

The cover features a close-up photograph of two beetles on a green leaf. One beetle is a spotted ladybug, and the other is a striped beetle. The background is a dark, blurred green.

CRC CRC Press
Taylor & Francis Group

Encyclopedia of
PEST MANAGEMENT
VOLUME II

Edited by
David Pimentel

Encyclopedia of
PEST MANAGEMENT
VOLUME II

Encyclopedias from Taylor & Francis Group

Agriculture Titles

Dekker Agropedia Collection (Eight-Volume Set)

ISBN: 0-8247-2194-2 13-Digit ISBN: 978-0-8247-2194-7

Encyclopedia of Agricultural, Food, and Biological Engineering

Edited by Dennis R. Heldman

ISBN: 0-8247-0938-1 13-Digit ISBN: 978-0-8247-0938-9

Encyclopedia of Animal Science

Edited by Wilson G. Pond and Alan Bell

ISBN: 0-8247-5496-4 13-Digit ISBN: 978-0-8247-5496-9

Encyclopedia of Pest Management

Edited by David Pimentel

ISBN: 0-8247-0632-3 13-Digit ISBN: 978-0-8247-0632-6

Encyclopedia of Plant and Crop Science

Edited by Robert M. Goodman

ISBN: 0-8247-0944-6 13-Digit ISBN: 978-0-8247-0944-0

Encyclopedia of Soil Science, Second Edition (Two-Volume Set)

Edited by Rattan Lal

ISBN: 0-8493-3830-1 13-Digit ISBN: 978-0-8493-3830-4

Encyclopedia of Water Science

Edited by B.A. Stewart and Terry Howell

ISBN: 0-8247-0948-9 13-Digit ISBN: 978-0-8247-0948-8

Business Titles

Encyclopedia of Library and Information Science, Second Edition (Four-Volume Set)

Edited by Miriam Drake

ISBN: 0-8247-2075-X 13-Digit ISBN: 978-0-8247-2075-9

Encyclopedia of Public Administration and Public Policy (Two-Volume Set)

Edited by Jack Rabin

ISBN: 0-8247-4748-8 13-Digit ISBN: 978-0-8247-4748-0

Chemistry Titles

Encyclopedia of Chromatography, Second Edition (Two-Volume Set)

Edited by Jack Cazes

ISBN: 0-8247-2785-1 13-Digit ISBN: 978-0-8247-2785-7

Encyclopedia of Surface and Colloid Science, Second Edition (Eight-Volume Set)

Edited by P. Somasundaran

ISBN: 0-8493-9615-8 13-Digit ISBN: 978-0-8493-9615-1

Chemistry Titles (cont'd)

Encyclopedia of Supramolecular Chemistry (Two-Volume Set)

Edited by Jerry L. Atwood and Jonathan W. Steed

ISBN: 0-8247-5056-X 13-Digit ISBN: 978-0-8247-5056-5

Engineering Titles

Encyclopedia of Chemical Processing (Five-Volume Set)

Edited by Sunggyu Lee

ISBN: 0-8247-5563-4 13-Digit ISBN: 978-0-8247-5563-8

Encyclopedia of Corrosion Technology, Second Edition

Edited by Philip A. Schweitzer, P.E.

ISBN: 0-8247-4878-6 13-Digit ISBN: 978-0-8247-4878-4

Encyclopedia of Energy Engineering and Technology (Three-Volume Set)

Edited by Barney L. Capehart

ISBN: 0-8493-3653-8 13-Digit ISBN: 978-0-8493-3653-9

Dekker Encyclopedia of Nanoscience and Nanotechnology (Five-Volume Set)

Edited by James A. Schwarz, Cristian I. Contescu, and Karol Putyera

ISBN: 0-8247-5055-1 13-Digit ISBN: 978-0-8247-5055-8

Encyclopedia of Optical Engineering (Three-Volume Set)

Edited by Ronald G. Driggers

ISBN: 0-8247-0940-3 13-Digit ISBN: 978-0-8247-0940-2

Coming Soon

Encyclopedia of Public Administration and Public Policy, Second Edition

Edited by Jack Rabin

ISBN: 1-4200-5275-6 13-Digit ISBN: 978-1-4200-5275-6

Encyclopedia of Water Science, Second Edition (Two-Volume Set)

Edited by Stanley W. Trimble

ISBN: 0-8493-9627-1 13-Digit ISBN: 978-0-8493-9627-4

Encyclopedia of Wireless and Mobile Communications (Three-Volume Set)

Edited by Borko Furht

ISBN: 1-4200-4326-9 13-Digit ISBN: 978-1-4200-4326-6

These titles are available both in print and online. To order, visit:

www.crcpress.com

Telephone: 1-800-272-7737

Fax: 1-800-374-3401

E-Mail: orders@crcpress.com

Encyclopedia of
PEST MANAGEMENT
VOLUME II

Edited by
David Pimentel
*Cornell University
Ithaca, New York*



CRC Press
Taylor & Francis Group
Boca Raton London New York

CRC Press is an imprint of the
Taylor & Francis Group, an **informa** business

CRC Press
Taylor & Francis Group
6000 Broken Sound Parkway NW, Suite 300
Boca Raton, FL 33487-2742

© 2007 by Taylor & Francis Group, LLC
CRC Press is an imprint of Taylor & Francis Group, an Informa business

No claim to original U.S. Government works
Printed in the United States of America on acid-free paper
10 9 8 7 6 5 4 3 2 1

International Standard Book Number-13: 978-1-4200-5361-6 (Hardcover)

This book contains information obtained from authentic and highly regarded sources. Reprinted material is quoted with permission, and sources are indicated. A wide variety of references are listed. Reasonable efforts have been made to publish reliable data and information, but the author and the publisher cannot assume responsibility for the validity of all materials or for the consequences of their use.

No part of this book may be reprinted, reproduced, transmitted, or utilized in any form by any electronic, mechanical, or other means, now known or hereafter invented, including photocopying, microfilming, and recording, or in any information storage or retrieval system, without written permission from the publishers.

For permission to photocopy or use material electronically from this work, please access www.copyright.com (<http://www.copyright.com/>) or contact the Copyright Clearance Center, Inc. (CCC) 222 Rosewood Drive, Danvers, MA 01923, 978-750-8400. CCC is a not-for-profit organization that provides licenses and registration for a variety of users. For organizations that have been granted a photocopy license by the CCC, a separate system of payment has been arranged.

Trademark Notice: Product or corporate names may be trademarks or registered trademarks, and are used only for identification and explanation without intent to infringe.

Visit the Taylor & Francis Web site at
<http://www.taylorandfrancis.com>

and the CRC Press Web site at
<http://www.crcpress.com>

Encyclopedia of Pest Management

David Pimentel

Editor

*Cornell University
Ithaca, New York, U.S.A.*

Editorial Advisory Board

Karen L. Bailey

*Agriculture and Agri-Food Canada, Saskatoon,
Saskatchewan, Canada*

George Ekström

Pesticide Control Consultant, Uppsala, Sweden

Diego O. Ferraro

*Universidad de Buenos Aires, Buenos Aires,
Argentina*

Geoff Gurr

*The University of Sydney, Orange,
New South Wales, Sydney*

Maurizio G. Paoletti

*Università di Padova via U. Bassi,
Padova, Italy*

Charles G. Summers

*Department of Entomology, University of
California, Davis & Kearney Agricultural
Center, Parlier, California, U.S.A.*

Joop van Lenteren

*Wageningen University and Research Centre,
Wageningen, The Netherlands*

Contributors

- Hector Achicanoy** / *Departamento de Agronomía, Universidad Nacional Colombia Sede Medellín, Medellín, Antioquia, Colombia*
- Ramon Albajes** / *Department of Entomology, Centre UDL-IRTA, University of Lleida, Lleida, Spain*
- Oscar Alomar** / *Department of Protecció Vegetal, IRTA, Cabrils (Barcelona), Spain*
- Árpád Ambrus** / *Agriculture and Biotechnology Laboratory, International Atomic Energy Agency, Vienna, Austria*
- Todd A. Anderson** / *Institute of Environmental and Human Health, Texas Tech University, Lubbock, Texas, U.S.A.*
- George Antonious** / *Water Quality Research, Kentucky State University, Frankfort, Kentucky, U.S.A.*
- Luis Felipe Arauz Cavallini** / *Escuela de Fitotecnia, Universidad de Costa Rica, San José, Costa Rica*
- John L. Aston** / *Safe Use Project, CropLife International, Nairobi, Kenya*
- Jesus Avilla** / *Centre UdL-IRTA for R+D, University of Lleida, Lleida, Catalonia, Spain*
- U. I. Baby** / *UPASI Tea Research Foundation, Valparai, Coimbatore, Tamil Nadu, India*
- Macario Bacilio Jimenez** / *CIBNOR, La Paz, Baja California Sur, Mexico*
- Rosalind Ball** / *Department of Plant Sciences, University of Saskatchewan, Saskatoon, Saskatchewan, Canada*
- Jane A.S. Barber** / *Mosquito Adulthood Section, College of Engineering Science Technology and Agriculture, Florida A&M University, Panama City, Florida, U.S.A.*
- H. Michael Barnes** / *Forest Products Laboratory, Mississippi State University, Mississippi State, Mississippi, U.S.A.*
- Diego Batlla** / *Departamento de Produccion Vegetal, Universidad de Buenos Aires, Buenos Aires, Argentina*
- Frederick P. Baxendale** / *Department of Entomology, University of Nebraska, Lincoln, Nebraska, U.S.A.*
- Frederick N. Bebe** / *Department of Nutrition and Health, Land Grant Program, Kentucky State University, Frankfort, Kentucky, U.S.A.*
- Roberto Benech-Arnold** / *C.O.N.I.C.E.T./Facultad de Agronomía, Universidad de Buenos Aires, Buenos Aires, Argentina*
- D. Michael Benson** / *Department of Plant Pathology, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- Walter J. Bentley** / *Kearney Agricultural Center, University of California, Statewide IPM Program, Parlier, California, U.S.A.*
- Nida Besbelli** / *International Programme on Chemical Safety, World Health Organization, Geneva, Switzerland*
- Alan R. Biggs** / *Kearneysville Tree Fruit Research and Education Center, West Virginia University, Kearneysville, West Virginia, U.S.A.*
- Susan Bjornson** / *Department of Biology, Saint Mary's University, Halifax, Nova Scotia, Canada*
- Noubar J. Bostanian** / *HRDC, Agriculture and Agri-Food Canada, St. Jean-sur-Richelieu, Quebec, Canada*
- Lars Olav Brandsæter** / *Department of Herbology, The Norwegian Crop Research Institute, Plant Protection Center, As, Norway*
- Luis Brenes** / *Escuela de Agronomía, Universidad de Costa Rica, San José, Costa Rica*

- Walter Brüsch** / *Department of Geochemistry, Geological Survey of Denmark and Greenland (GEUS), Copenhagen K., Denmark*
- Tibor Bukovinszky** / *Department of Plant Sciences, Laboratory of Entomology, Wageningen University and Research Centre, Wageningen, The Netherlands*
- László Bura** / *Central Service for Plant Protection and Soil Conservation, Budapest, Hungary*
- Giovanni Burgio** / *Dipartimento di Scienze e Tecnologie Agroambientali-Entomologia, Alma Mater Studiorum Università di Bologna, Bologna, Italy*
- Eileen A. Buss** / *Entomology and Nematology Department, University of Florida, Gainesville, Florida, U.S.A.*
- G. Butchaiah** / *National Biotechnology Center, Rajiv Gandhi College of Veterinary and Animal Sciences, Pondicherry, Pondicherry, India*
- Agneta Sundén Byléhn** / *UNEP Chemicals, United Nations Environment Program, Chatelaine (Geneva), Switzerland*
- Héctor A. Cárcamo** / *Lethbridge Research Centre, Agriculture and Agri-Food Canada, Lethbridge, Alberta, Canada*
- Ring Carde** / *Department of Entomology, University of California, Riverside, California, U.S.A.*
- Hugo Cerda-Perez** / *Simon Rodriguez University, Caracas, Venezuela*
- Fabio Chaverri** / *Central American Institute for Research on Toxic Substances (IRET), Universidad Nacional, Heredia, Costa Rica*
- Maria V. Cilveti** / *Department of Entomology, College of Agriculture and Life Sciences Cornell University, Ithaca, New York, U.S.A.*
- Christopher A. Clark** / *Department of Plant Pathology and Crop Physiology, Louisiana State University, Baton Rouge, Louisiana, U.S.A.*
- John S. Clayton** / *National Institute of Water and Atmospheric Research, Hamilton, New Zealand*
- Joel R. Coats** / *Department of Entomology, Iowa State University, Ames, Iowa, U.S.A.*
- Harold Coble** / *Department of Crop Science, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- Bruce J. Cochrane** / *Biology Department, University of South Florida, Tampa, Florida, U.S.A.*
- Rosemary H. Collier** / *Warwick HRI, The University of Warwick, Wellesbourne, Warwick, U.K.*
- Claudio Colosio** / *International Centre for Pesticide Safety, Busto Garolfo, Italy*
- Joseph D. Cornell** / *College of Environmental Science and Forestry, State University of New York, Syracuse, New York, U.S.A.*
- Phil Cowan** / *Department of Vertebrate Pest Ecology, Landcare Research New Zealand, Ltd., Palmerston North, New Zealand*
- C. C. Craig Jr.** / *Department of Plant Sciences, University of Tennessee, Jackson, Tennessee, U.S.A.*
- Marc A. Cubeta** / *Center for Integrated Fungal Research, Plant Pathology, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- Thomas W. Culliney** / *Center for Plant Health Science and Technology, USDA, APHIS, PPQ, Raleigh, North Carolina, U.S.A.*
- Kent M. Daane** / *Department of Environmental Science, Policy, and Management, Kearney Agricultural Center, University of California-Berkeley, Berkeley, California, U.S.A.*
- Margery Daughtrey** / *Department of Plant Pathology, Cornell University, Long Island Horticultural Research and Extension Center, Riverhead, New York, U.S.A.*
- Maria Elita Batista de Castro** / *Embrapa Recursos Genéticos e Biotecnologia, Brasilia, Brazil*
- Patrick De Clerq** / *Department of Crop Protection, Laboratory of Agrozoology, Faculty of Agricultural and Applied Biological Sciences, Ghent University, Ghent, Belgium*
- Richard de Shazo** / *University of Mississippi Medical Center, Jackson, Mississippi, U.S.A.*
- Marlinda Lobo de Souza** / *Embrapa Recursos Genéticos e Biotecnologia, Brasília, Brazil*
- Barbara Dinham** / *Eurolink Centre, Pesticide Action Network UK, London, U.K.*
- J. K. Dubey** / *Department of Entomology, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India*

- Raymond-Marie Duchesne** / *Direction de l'Environnement et du Développement Durable, Quebec Ministry of Agriculture, Fisheries, and Food, Sainte-Foy, Quebec, Canada*
- George Ekström** / *Swedish National Chemicals Inspectorate (KEMI), Solna, Sweden*
- James F. Ellerhoff** / *Iowa Department of Agriculture and Land Stewardship, Des Moines, Iowa, U.S.A.*
- Kathleen Fagerstone** / *APHIS/WS, USDA – National Wildlife Research Center, Fort Collins, Colorado, U.S.A.*
- Gitte Felding** / *Geological Survey of Denmark and Greenland (GEUS), Copenhagen K., Denmark*
- Alessandra Ferrandino** / *Dipartimento di Coltura Arborea, Universita di Torino, Grugliasco, Italy*
- Diego O. Ferraro** / *IFEVA, Departamento de Recursos Naturales y Ambiente, Universidad de Buenos Aires—CONICET, Buenos Aires, Argentina*
- David N. Ferro** / *Department of Entomology, University of Massachusetts, Amherst, Massachusetts, U.S.A.*
- Stan Finch** / *Warwick HRI, The University of Warwick, Wellesbourne, Warwick, U.K.*
- J. Howard Frank** / *Department of Entomology and Nematology, University of Florida, Gainesville, Florida, U.S.A.*
- Eugene L. Fytche** / *Fytche Enterprises, Almonte, Ontario, Canada*
- Ján Gallo** / *Department of Plant Protection, Faculty of Agronomy, Slovak Agricultural University, Nitra, Slovak Republic*
- Abraham Gamliel** / *Department of Agricultural and Environmental Engineering, Laboratory of Pest Management Application, ARO Volcani Center, Bet Dagan, Israel*
- Vincent F. Garry** / *Department of Laboratory Medicine and Pathology, University of Minnesota, Minneapolis, Minnesota, U.S.A.*
- Jacques Gill** / *Department of Soil Science and Agri-Food Engineering, College of Agriculture and Food Sciences, University of Laval, Quebec, Quebec, Canada*
- Ashok P. Giri** / *Plant Molecular Biology Unit, Division of Biochemical Sciences, National Chemical Laboratory, Pune, Maharastra, India*
- Stephen R. Gliessman** / *Department of Environmental Studies, University of California-Santa Cruz, Santa Cruz, California, U.S.A.*
- Jerome Goddard** / *Mississippi Department of Health, Jackson, Mississippi, U.S.A.*
- Peter B. Goodell** / *University of California, Davis, California, U.S.A.*
- Bruce D. Gossen** / *Saskatoon Research Centre, Agriculture and Agri-Food Canada, Saskatoon, Saskatchewan, Canada*
- Edward J. Grafius** / *Department of Entomology, Michigan State University, East Lansing, Michigan, U.S.A.*
- Simon Grenier** / *Biologie Fonctionnelle Insectes et Interactions (BF21), UMR INRA/INSA de Lyon, Villeurbanne Cedex, France*
- Sylvia Guidoni** / *Dipartimento di Coltura Arborea, Universita di Torino, Grugliasco, Italy*
- Tom Gulya** / *Northern Crop Science Lab, United States Department of Agriculture, Agricultural Research Service, Fargo, North Dakota, U.S.A.*
- Divender Gupta** / *Department of Entomology, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India*
- Vidya S. Gupta** / *Plant Molecular Biology Unit, Division of Biochemical Sciences, National Chemical Laboratory, Pune, Maharashtra, India*
- C. O. Gwathmey** / *Department of Plant Sciences, University of Tennessee, Jackson, Tennessee, U.S.A.*
- Ann E. Hajek** / *Department of Entomology, Cornell University, Ithaca, New York, U.S.A.*
- Lise Stengård Hansen** / *Danish Pest Infestation Laboratory, Danish Institute of Agricultural Sciences, Kgs. Lyngby, Denmark*
- Marvin K. Harris** / *Department of Entomology, Texas A&M University, College Station, Texas, U.S.A.*
- Glen L. Hartman** / *Agricultural Research Service, United States Department of Agriculture and University of Illinois, Urbana, Illinois, U.S.A.*
- John S. Hartung** / *Plant Germplasm Quarantine Office, United States Department of Agriculture – Agricultural Research Service, Beltsville, Maryland, U.S.A.*

- James D. Harwood** / *Department of Entomology, University of Kentucky, Lexington, Kentucky, U.S.A.*
- Harlene Hatterman-Valenti** / *Plant Sciences Department, North Dakota State University, Fargo, North Dakota, U.S.A.*
- Zane R. Helsel** / *Department of Extension Specialists, Cook College, Rutgers University, New Brunswick, New Jersey, U.S.A.*
- Thomas J. Henneberry** / *Arid Land Agricultural Research Center, USDA-ARS, Maricopa, Arizona, U.S.A.*
- Nancy Hinkle** / *Department of Entomology, University of Georgia, Athens, Georgia, U.S.A.*
- E. Richard Hoebeker** / *Department of Entomology, Cornell University, Ithaca, New York, U.S.A.*
- Ary A. Hoffman** / *Department of Zoology, Cooperative Research Centre for Viticulture (CRCV), Glen Osmond, South Australia, Australia*
- Eric J. Hoffman** / *Michigan State University, East Lansing, Michigan, U.S.A.*
- Mike Hoffmann** / *Department of Entomology, Cornell University, Ithaca, New York, U.S.A.*
- Derek Hollomon** / *Department of Agricultural Sciences, Long Ashton Research Station, Institute of Arable Crops Research, University of Bristol, Bristol, U.K.*
- David J. Horn** / *Department of Entomology, Ohio State University, Columbus, Ohio, U.S.A.*
- Joyce S. Hornstein** / *Department of Entomology, Iowa State University, Ames, Iowa, U.S.A.*
- David R. Horton** / *USDA-ARS, Wapato, Washington, U.S.A.*
- F. W. Howard** / *Fort Lauderdale Research and Education Center, University of Florida, Fort Lauderdale, Florida, U.S.A.*
- Nguyen Huu Huan** / *Department of Plant Protection, Ministry of Agriculture and Rural Development, Ho Chi Minh City, Vietnam*
- Chau-Chin Hung** / *Taiwan Agricultural Chemicals and Toxic Substances Research Institute, Council of Agriculture, Executive Yuan, P. R. China*
- Jenn-Sheng Hwang** / *Department of Applied Toxicology, Taiwan Agricultural Chemicals and Toxic Substances Research Institute, Council of Agriculture, Taiwan, Republic of China-Taiwan*
- Sheau-Fang Hwang** / *Alberta Research Council, Vegreville, Alberta, Canada*
- Ana Iglesias** / *Goddard Institute for Space Studies, New York, New York, U.S.A.*
- Francisco Infante** / *El Colegio de la Frontera Sur (ECOSUR), Tapachula, Chiapas, Mexico*
- Joseph Ingerson-Mahar** / *Rutgers University, New Brunswick, New Jersey, U.S.A.*
- Hitoshi Ito** / *Takasaki Radiation Chemistry Research Establishment, Japan Atomic Energy Research Institute, Takasaki, Gunma, Japan*
- Guido Jach** / *Max-Planck Institute for Plant Breeding Research, Cologne, Germany*
- Marshall W. Johnson** / *Department of Entomology, University of California-Riverside, Riverside, California, U.S.A.*
- Sushil Kumar Kabra** / *Department of Pediatrics, All India Institute of Medical Sciences, New Delhi, India*
- Masashi Kakizaki** / *Hokkaido Ornamental Plants and Vegetables Research Center, Hokkaido, Japan*
- Sylvia I. Karlsson** / *Finland Futures Research Centre, Turku School of Economics and Business Administration, Tampere, Finland*
- Anthony P. Keinath** / *Coastal Research and Education Center, Clemson University, Charleston, South Carolina, U.S.A.*
- Deepak Raj Khajuria** / *Regional Horticultural Research Station, Dr. Y.S. Parmar University of Horticulture and Forestry, Kullu, Himachal Pradesh, India*
- Mohamed Khelifi** / *Department of Soil Science and Agri-Food Engineering, Université Laval, Sainte-Foy, Quebec, Canada*
- Gurdev S. Khush** / *International Rice Research Institute, Davis, California, U.S.A.*
- Daniel L. Kline** / *CMAVE, United States Department of Agriculture (USDA-ARS), Gainesville, Florida, U.S.A.*
- Philip Koehler** / *Entomology and Nematology Department, University of Florida, Gainesville, Florida, U.S.A.*

- Stephen R. Koenning** / *Department of Plant Pathology, College of Agriculture and Life Sciences, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- Steven T. Koike** / *UC Cooperative Extension, Salinas, California, U.S.A.*
- Livia Nemeth Konda** / *Analytical Chemistry Department, Institute for Veterinary Medicinal Products, Budapest, Hungary*
- Albrecht M. Koppenhöfer** / *Department of Entomology, Cook College, Rutgers University, New Brunswick, New Jersey, U.S.A.*
- Joseph Kovach** / *Department of Entomology, IPM Program, Ohio State University, Wooster, Ohio, U.S.A.*
- Mirco Kreibich** / *Division 402: Regional Development Banks, IFAD, Federal Ministry for Economic Cooperation and Development, Bonn, Germany*
- N. Pradeep Kumar** / *Vector Control Research Centre (Indian Council of Medical Research), Pondicherry, Pondicherry, India*
- Henrik Kylin** / *Swedish University of Agricultural Sciences, Uppsala, Sweden*
- Claude Laguë** / *College of Engineering, University of Saskatchewan, Saskatoon, Saskatchewan, Canada*
- David B. Langston Jr.** / *Rural Development Center, University of Georgia, Tifton, Georgia, U.S.A.*
- Alberto Lanzoni** / *Dipartimento di Scienze e Tecnologie Agroambientali-Entomologia, Alma Mater Studiorum Università di Bologna, Bologna, Italy*
- Patricia S. Larráin** / *Instituto de Investigaciones Agropecuarias, Centro Regional de Investigación INIA-Intihuasi, La Serena, Chile*
- Barbra C. Larson** / *Department of Environmental Sciences, Institute of Food and Agricultural Sciences, University of Florida, Gainesville, Florida, U.S.A.*
- Kirk D. Larson** / *Department of Plant Sciences, University of California-Davis, Davis, California, U.S.A.*
- John LeBoeuf** / *AgriDataSensing, Inc., Fresno, California, U.S.A.*
- Hugh Lehman** / *University of Guelph, Guelph, Ontario, Canada*
- Norman C. Leppla** / *Department of Entomology and Nematology, Institute of Food and Agricultural Sciences, University of Florida, Gainesville, Florida, U.S.A.*
- Nan Lin** / *Department of Laboratory Medicine and Pathology, University of Minnesota, Minneapolis, Minnesota, U.S.A.*
- David Lockwood** / *Department of Plant Sciences, University of Tennessee, Knoxville, Tennessee, U.S.A.*
- Rakesh Lodha** / *Department of Pediatrics, All India Institute of Medical Sciences, New Delhi, India*
- Leslie London** / *Occupational and Environmental Health Research Unit, School of Public Health and Primary Health Care, University of Cape Town, Observatory, Western Cape, South Africa*
- E. Rolando Lopez-Gutierrez** / *Clemson University, Charleston, South Carolina, U.S.A.*
- Alan MacNicoll** / *Department for Environment, Food and Rural Affairs, Central Science Laboratories, York, U.K.*
- Antonio Masetti** / *Dipartimento di Scienze e Tecnologie Agroambientali-Entomologia, Alma Mater Studiorum Università di Bologna, Bologna, Italy*
- Gloria S. McCutcheon** / *Clemson University, Charleston, South Carolina, U.S.A.*
- James R. Miller** / *Michigan State University, East Lansing, Michigan, U.S.A.*
- Jeffrey P. Mitchell** / *Kearney Agricultural Center, University of California, Parlier, California, U.S.A.*
- Charles L. Mohler** / *Department of Crop and Soil Sciences, College of Agriculture and Life Sciences, Cornell University, Ithaca, New York, U.S.A.*
- Flavio Moscardi** / *Embrapa Soja, Brazil*
- Shannon C. Mueller** / *University of California, Davis, California, U.S.A.*
- Paul Mugge** / *Sutherland, Iowa, U.S.A.*
- Krishnoji Rao Muktha Bai** / *Food Protectants and Infestation Control Department, Central Food Technological Research Institute, Mysore, Karnataka, India*
- Heinz Müller-Schaerer** / *Département de Biologie/Ecologie, Université de Fribourg/Perolles, Fribourg, Switzerland*

- N. Muraleedharan** / *UPASI Tea Research Foundation, Nirar Dam BPO, Valparai, Coimbatore, Tamil Nadu, India*
- Joji Muramoto** / *Center for Agroecology and Sustainable Food Systems, University of California-Santa Cruz, Santa Cruz, California, U.S.A.*
- Larry L. Murdock** / *Department of Entomology, Purdue University, West Lafayette, Indiana, U.S.A.*
- Douglas L. Murray** / *Department of Sociology, Colorado State University, Fort Collins, Colorado, U.S.A.*
- Paul Neve** / *Western Australian Herbicide Resistance Initiative (WAHRI), University of Western Australia (UWA), Crawley, Western Australia, Australia*
- Aiwerasia V.F. Ngowi** / *Pesticide Environmental Management Centre, Tropical Pesticides Research Institute, Arusha, Arusha, Tanzania*
- Leonard Nunney** / *Department of Biology, University of California-Riverside, Riverside, California, U.S.A.*
- John J. Obrycki** / *Department of Entomology, University of Kentucky, Lexington, Kentucky, U.S.A.*
- E.-C. Oerke** / *Institute for Plant Diseases, Rheinische Friedrich-Wilhelms-Universitaet Bonn, Bonn, Germany*
- Philip Oduor Owino** / *Department of Botany, University of Kenyatta, Nairobi, Kenya*
- Margareta Palmborg** / *Swedish Poisons Information Centre, Stockholm, Sweden*
- Myna Panemangalore** / *Department of Nutrition and Health, Land Grant Program, Kentucky State University, Frankfort, Kentucky, U.S.A.*
- Alberto Pantoja** / *Agricultural Research Service, Subarctic Agricultural Research Unit, United States Department of Agriculture, Fairbanks, Alaska, U.S.A.*
- Maurizio G. Paoletti** / *Dipartimento di Biologia, Universita di Padova, Padova, Italy*
- S. K. Patyal** / *Department of Entomology and Apiculture, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India*
- Jorge E. Peña** / *Tropical Research and Education Center, University of Florida, Homestead, Florida, U.S.A.*
- Hans Persson** / *Swedish Poisons Information Centre, Stockholm, Sweden*
- David Pimentel** / *Department of Entomology, College of Agriculture and Life Sciences Cornell University, Ithaca, New York, U.S.A.*
- Arevik Poghosyan** / *CIBNOR, La Paz, Baja California Sur, Mexico*
- John Pontius** / *Training Specialist, The FAO Programme for Community IPM in Asia, Jakarta, Indonesia*
- Francisco Posada** / *Insect Biocontrol Laboratory, United States Department of Agriculture, Agricultural Research Service, Beltsville, Maryland, U.S.A.*
- Stephen Powles** / *Department of Agriculture, Western Australia Herbicide Resistance Initiative (WAHRI), University of Western Australia (UWA), Crawley, Western Australia, Australia*
- Nilima Prabhaker** / *Western Cotton Res Lab, University of California, Riverside, Phoenix, Arizona, U.S.A.*
- Jenny Pronczuk** / *International Programme on Chemical Safety, World Health Organization, Geneva, Switzerland*
- Gilbert Proulx** / *Alpha Wildlife Research and Management Ltd., Sherwood Park, Alberta, Canada*
- Carlos E. Quiroz** / *Instituto de Investigaciones Agropecuarias, Centro Regional de Investigación INIA-Intihuasi, La Serena, Chile*
- Bruce Radford** / *Department of Natural Resources, Bileola Research Station, Bileola, Australia*
- Rudraraju A. Raju** / *Agricultural Research Station, A.R. Andhra Pradesh Agricultural University (AR APAU), Maruteru, Hyderabad, India*
- Carolyn J. Randall** / *Pesticide Education Program, Michigan State University, East Lansing, Michigan, U.S.A.*
- C. N. Rao** / *National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India*
- Michael J. Raupp** / *Department of Entomology, College of Life Sciences, University of Maryland, College Park, Maryland, U.S.A.*

- Randall C. Reeder** / *Food, Agricultural and Biological Engineering Department, The Ohio State University, Columbus, Ohio, U.S.A.*
- Robert E. Reichard** / *Scientific and Technical Department, World Organization for Animal Health (OIE), Paris, France*
- Landon H. Rhodes** / *Department of Plant Pathology, The Ohio State University, Columbus, Ohio, U.S.A.*
- Helmut Riedl** / *Mid-Columbia Agricultural Research and Extension Center, Oregon State University, Hood River, Oregon, U.S.A.*
- David F. Ritchie** / *Department of Plant Pathology, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- William H. Robinson** / *Urban Pest Control Research, Christiansburg, Virginia, U.S.A.*
- Cynthia Rosenzweig** / *Columbia University, Goddard Institute for Space Studies, New York, New York, U.S.A.*
- Amy Y. Rossman** / *United States Department of Agriculture, Agricultural Research Service, Beltsville, Maryland, U.S.A.*
- Hanna Andrea Rother** / *Occupational and Environmental Health Research Unit, School of Public Health and Family Medicine, University of Cape Town, Observatory, South Africa*
- Maristella Rubbiani** / *Laboratorio di Tossicologia Applicata, Istituto Superiore de Sàntita, Rome, Italy*
- John Ruberson** / *Department of Entomology, University of Georgia, Tifton, Georgia, U.S.A.*
- Clemens Ruepert** / *Central American Institute for Research on Toxic Substances (IRET), Universidad Nacional, Heredia, Costa Rica*
- Findlay E. Russell** / *Department of Pharmacology and Toxicology, University of Arizona, Tucson, Arizona, U.S.A.*
- Stuart Rutherford** / *Crop Biology Resource Centre, SA Sugarcane Research Institute, Mount Edgecombe, Kwa-Zulu Natal, South Africa*
- Alarik Sandrup** / *Ministry of Agriculture, Stockholm, Sweden*
- Michael E. Scharf** / *Department of Entomology, Purdue University, West Lafayette, Indiana, U.S.A.*
- Michael Schauff** / *United States Department of Agriculture, Agricultural Research Service, Beltsville, Maryland, U.S.A.*
- Ulrich Schlottmann** / *Head of Division IG II 3, Federal Ministry for the Environment, and Nature Conservation and Nuclear Safety, Bonn, Germany*
- Graham Scoles** / *Department of Plant Sciences, University of Saskatchewan, Saskatoon, Saskatchewan, Canada*
- Vineet K. Sharma** / *Department of Plant Pathology, Punjab Agricultural University, Ludhiana, Punjab, India*
- Douglas V. Shaw** / *Department of Plant Sciences, University of California-Davis, Davis, California, U.S.A.*
- Barbara B. Shew** / *Department of Plant Pathology, North Carolina State University, Raleigh, North Carolina, U.S.A.*
- V. J. Shivankar** / *Department of Entomology, National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India*
- Paula M. Shrewsbury** / *Department of Entomology, College of Life Sciences, University of Maryland, College Park, Maryland, U.S.A.*
- Rudra Singh** / *Potato Research Centre, Agriculture and Agri-Food Canada, Fredericton, New Brunswick, Canada*
- Shyam Singh** / *National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India*
- Tanu Singhal** / *Department of Pediatrics, All India Institute of Medical Sciences, New Delhi, India*
- William Smith** / *Department of Entomology, Cornell University, Ithaca, New York, U.S.A.*
- Daniele Sommaggio** / *Biostudio, Velo d'Astico (VI), Italy*
- Punnee Soonthornpoch** / *Department of Natural Sciences, Blinn College, Bryan, Texas, U.S.A.*
- Juliana J. Soroka** / *Saskatoon Research Centre, Agriculture and Agri-Food Canada, Saskatoon, Saskatchewan, Canada*

- Eric B. Spurr** / *Department of Wildlife Ecology, Landcare Research New Zealand, Ltd., Lincoln, New Zealand*
- James J. Stapleton** / *Statewide IPM Project, UC Kearney Agricultural Center, Parlier, California, U.S.A.*
- Richard Stouthamer** / *Department of Entomology, University of California, Riverside, California, U.S.A.*
- Evamarie Straube** / *Institute of Occupational Medicine, University of Greifswald, Greifswald, Germany*
- Sebastian Straube** / *Department of Physiology, Laboratory of Molecular and Cellular Signalling, University of Oxford, Oxford, U.K.*
- Wolfgang Straube** / *Department of Gynecology and Obstetrics, University of Greifswald, Greifswald, Germany*
- Krishna V. Subbarao** / *Department of Plant Pathology, (U.S. Agricultural Research Station), University of California at Davis, Salinas, California, U.S.A.*
- R. Mark Sulc** / *Department of Horticulture and Crop Science, The Ohio State University, Columbus, Ohio, U.S.A.*
- Charles G. Summers** / *Department of Entomology, University of California, Parlier, California, U.S.A.*
- George W. Sundin** / *Department of Plant Pathology, Michigan State University, East Lansing, Michigan, U.S.A.*
- Vaijayanti A. Tamhane** / *Plant Molecular Biology Unit, Division of Biochemical Sciences, National Chemical Laboratory, Pune, Maharashtra, India*
- Peter L. Taylor** / *Department of Sociology, Colorado State University, Fort Collins, Colorado, U.S.A.*
- T. S. Thind** / *Department of Plant Pathology, Punjab Agricultural University, Ludhiana, Punjab, India*
- Linda J. Thomson** / *Centre for Environmental Stress and Adaptation Research (CESAR), University of Melbourne, Parkville, Victoria, Australia*
- Vicki Tolmay** / *Small Grain Institute, Agricultural Research Council, Bethlehem, South Africa*
- Avinash M. Tope** / *Department of Nutrition and Health, Land Grant Program, Kentucky State University, Frankfort, Kentucky, U.S.A.*
- Nick C. Toscano** / *Department of Entomology, University of California, Riverside, California, U.S.A.*
- Pasquale Trematerra** / *Department SAVA, University of Molise, Campobasso, Italy*
- Thomas R. Unruh** / *USDA-ARS, Wapato, Washington, U.S.A.*
- Arnold van Huis** / *Laboratory of Entomology, Wageningen University, Wageningen, The Netherlands*
- Joop C. van Lenteren** / *Department of Plant Sciences, Laboratory of Entomology, Wageningen University and Research Centre, Wageningen, The Netherlands*
- P.C.J. van Rijn** / *Center for Terrestrial Ecology, Netherlands Institute of Ecology (NIOO-KNAW), Heteren, The Netherlands*
- Lucia Varela** / *University of California Cooperative Extension, Sonoma County, Santa Rosa, California, U.S.A.*
- Fernando E. Vega** / *Insect Biocontrol Laboratory, United States Department of Agriculture, Agricultural Research Service, Beltsville, Maryland, U.S.A.*
- L.E.M. Vet** / *Department of Multitrophic Interactions, Netherlands Institute of Ecology (NIOO-KNAW), Heteren, The Netherlands*
- Martin Vila-Aiub** / *School of Plant Biology, Faculty of Natural and Agricultural Sciences, Western Australian Herbicide Resistance Initiative (WAHRI), University of Western Australia (UWA), Crawley, Western Australia, Australia*
- Felix L. Wäckers** / *Center for Terrestrial Ecology, Netherlands Institute of Ecology (NIOO-KNAW), Heteren, The Netherlands*
- Farid Waliyar** / *ICRISAT Patancheru P.O., Hyderabad, Andhra Pradesh, India*
- Edward D. Walker** / *Michigan State University, East Lansing, Michigan, U.S.A.*
- Stephen C. Weller** / *Department of Horticulture and Landscape Architecture, Purdue University, West Lafayette, Indiana, U.S.A.*
- Rohan D.S. Wells** / *National Institute of Water and Atmospheric Research, Hamilton, New Zealand*

- Catharina Wesseling** / *Central American Institute for Studies on Toxic Substances (IRET),
Universidad Nacional, Heredia, Costa Rica*
- Gerald E. Wilde** / *Department of Entomology, Kansas State University, Manhattan, Kansas, U.S.A.*
- Anne Wilson** / *Department of Entomology, College of Agriculture and Life Sciences,
Cornell University, Ithaca, New York, U.S.A.*
- Alexandra Wilson-Rummenie** / *Department of Natural Resources, Biloela Research Station,
Biloela, Australia*
- Alemayehu Wodageneh** / *Prevention and Disposal, Obsolete Pesticide Stocks, Food and Agricultural
Organization of the United Nations (FAO), Rome, Italy*
- Alvin R. Womac** / *Department of Agricultural and Biosystems Engineering, University of Tennessee,
Knoxville, Tennessee, U.S.A.*
- Denis J. Wright** / *Department of Biological Sciences, Imperial College London, Ascot, U.K.*
- Mark G. Wright** / *Plant and Environmental Protection Sciences, University of Hawaii at Manoa,
Honolulu, Hawaii, U.S.A.*
- Frank G. Zalom** / *Department of Entomology, University of California-Davis, Davis,
California, U.S.A.*
- Gina Holguin Zehfuss** / *CIBNOR, La Paz, Baja California Sur, Mexico*
- He Zhong** / *Public Health Entomology Research and Education Center, Florida A&M University,
Panama City, Florida, U.S.A.*
- Keyan Zhu-Salzman** / *Department of Entomology, Texas A&M University, College Station,
Texas, U.S.A.*
- Lewis H. Ziska** / *Crop Systems and Global Change Laboratory, United States Department of
Agriculture (USDA-ARS), Beltsville, Maryland, U.S.A.*

Contents

| | |
|---|-------|
| <i>Contributors</i> | vii |
| <i>Preface</i> | xxiii |
| <i>About the Editor</i> | xxv |
| Adjuvants and Carriers / <i>Nan Lin and Vincent F. Garry</i> | 1 |
| Aerial Ultra-Low-Volume Application of Insecticide to Control Adult Mosquitoes: | |
| Minimizing Non-Target Impacts / <i>He Zhong</i> | 4 |
| Alfalfa Diseases: Ecology and Control / <i>Landon H. Rhodes and R. Mark Sulc</i> | 7 |
| Alfalfa Insects: Ecology and Management / <i>John J. Obrycki and James D. Harwood</i> | 11 |
| Animal Breeding / <i>G. Butchayah</i> | 14 |
| Aphid and Aphid-Borne Virus Management: Squash Disease Control / <i>Charles G. Summers, James J. Stapleton and Jeffrey P. Mitchell</i> | 17 |
| Asian Longhorned Beetle: Ecology and Control / <i>Ann E. Hajek</i> | 21 |
| Asian Longhorned Beetle: Invasion on North American Urban Forests / <i>E. Richard Hoebeke</i> | 25 |
| Bacterial Pest Controls / <i>David N. Ferro</i> | 30 |
| Biocontrol: Limits to Use / <i>Joseph D. Cornell</i> | 33 |
| Bioindicators: Use for Assessing Sustainability of Farming Practices / <i>Joji Muramoto and Stephen R. Gliessman</i> | 37 |
| Biological Control of Stored-Product Pests / <i>Lise Stengård Hansen</i> | 42 |
| Biotechnology / <i>Maurizio G. Paoletti</i> | 45 |
| Bird Control Chemicals / <i>Eric B. Spurr</i> | 52 |
| Cabbage Diseases: Ecology and Control / <i>Anthony P. Keinath, Marc A. Cubeta and David B. Langston Jr.</i> | 56 |
| Cereals: Growing Systems and Pest Occurrence / <i>Ján Gallo</i> | 60 |
| Chemigation / <i>Patricia S. Larrain and Carlos E. Quiroz</i> | 63 |
| Chemistry of Pesticides / <i>J. K. Dubey and S. K. Patyal</i> | 67 |
| Cherry Diseases: Ecology and Control / <i>George W. Sundin</i> | 75 |
| Cherry Insects: Ecology and Control / <i>Helmut Riedl and Jesus Avilla</i> | 79 |
| Climate and Pest Outbreaks / <i>Ana Iglesias and Cynthia Rosenzweig</i> | 87 |
| Coconut Insects: Ecology and Control / <i>F. W. Howard</i> | 90 |
| Coffee Insects: Ecology and Control / <i>Fernando E. Vega, Francisco Posada and Francisco Infante</i> | 95 |
| Colorado Potato Beetle: Thermal Control / <i>Mohamed Khelifi, Raymond-Marie Duchesne, Claude Laguë and Jacques Gill</i> | 99 |
| Consumer Concerns about Pesticides and Pests / <i>George Ekström and Margareta Palmberg</i> | 102 |
| Cowpea Insects: Ecology and Control / <i>Keyan Zhu-Salzman and Larry L. Murdock</i> | 106 |
| Crop Insect Control: Ant Roles / <i>E. Rolando Lopez-Gutierrez and Gloria S. McCutcheon</i> | 109 |
| Crop Insect Control: Red Imported Fire Ants / <i>Gloria S. McCutcheon and John Ruberson</i> | 113 |
| Crop Losses to Animal Pests, Plant Pathogens, and Weeds / <i>E.-C. Oerke</i> | 116 |
| Crop Residues and Pest Problems / <i>Bruce Radford and Alexandra Wilson-Rummenie</i> | 121 |
| Crop Rotations for Weed Control / <i>Paul Mugge</i> | 124 |

| | |
|---|-----|
| Cross-Resistance to Pesticides / Bruce J. Cochrane | 128 |
| Cruciferous Root Crop Insects: Ecology and Control / Stan Finch and Rosemary H. Collier | 131 |
| Defoliants for Cotton / C. O. Gwathmey and C. C. Craig Jr. | 135 |
| Delusory Parasitosis / Nancy Hinkle | 138 |
| Dengue / Sushil Kumar Kabra, Tanu Singhal and Rakesh Lodha | 141 |
| Developing Cooperations: Pest and Pesticide Management / George Ekström, Barbara Dinham and Henrik Kylin | 144 |
| Domestication of Agricultural Crops / Graham Scoles and Rosalind Ball | 150 |
| Energy Cost and Use in Pesticide Production / Diego O. Ferraro | 153 |
| Energy in Pesticide Production and Use / Zane R. Helsel | 157 |
| Enhanced Microbial Degradation of Pesticides / Todd A. Anderson and Joel R. Coats | 161 |
| Equipment for Ground Applications to Adult Mosquitoes / Jane A.S. Barber | 163 |
| Ethics, Biotechnology, and Pesticides / Hugh Lehman | 166 |
| Evolved Herbicide Resistance: Fitness Costs / Martin Vila-Aiub, Paul Neve and Stephen Powles | 169 |
| Facultative Predation as a Biological Control / Oscar Alomar | 172 |
| Field Release, Captive Rearing for / Leonard Nunney | 175 |
| Filariasis / N. Pradeep Kumar | 179 |
| Fire Ant Attacks on Humans and Animals / Jerome Goddard and Richard de Shazo | 183 |
| Flooding: Physiological Adaptations and Weed Control / Rudraraju A. Raju | 185 |
| Fruit Crop Pest Management: Weeds / Sylvia Guidoni and Alessandra Ferrandino | 190 |
| Fumigation / Abraham Gamliel | 194 |
| Genetics of Resistance and Plant Breeding / Guido Jach | 197 |
| Grape Production in Australia: Integrated Strategies and Bioindicators for Sustainability / Linda J. Thomson and Ary A. Hoffman | 200 |
| Grapes and Insects: Ecology and Control / Walter J. Bentley, Lucia Varela and Kent M. Daane | 207 |
| Greenhouse Plant Pathogens / Margery Daughtrey | 214 |
| Groundwater, Pesticides in / Walter Brüsch and Gitte Felding | 218 |
| Hazard Labeling / Leslie London and Hanna Andrea Rother | 223 |
| Health Impacts in Developing Countries / Aiwerasia V.F. Ngowi, Catharina Wesseling and Leslie London | 228 |
| <i>Helicoverpa armigera</i>: Ecology and Control Using Novel Biotechnological Approaches / Vaijayanti A. Tamhane, Ashok P. Giri and Vidya S. Gupta | 232 |
| Hormonal Disruption in Humans / Evamarie Straube, Sebastian Straube and Wolfgang Straube | 237 |
| Host-Plant Selection by Insects / Rosemary H. Collier and Stan Finch | 240 |
| Household Pest Management: Insects and Mites / William H. Robinson | 244 |
| Hoverflies: Indicators of Sustainable Farming and Potential Control of Aphids / Daniele Sommaggio and Giovanni Burgio | 247 |
| Immune Deficiency Effects / Claudio Colosio | 251 |
| Insect Pest Dispersal / V. J. Shivankar, C. N. Rao and Shyam Singh | 255 |
| Insect Pest Management / Thomas J. Henneberry | 258 |
| Insect Pest Management: Lawns / Frederick P. Baxendale | 261 |
| Insecticide Reduction on Lawns / Eileen A. Buss and Philip Koehler | 267 |
| Insecticide Resistance Management / Edward J. Grafius | 271 |
| Integrated Pest Management: Principles with Emphasis on Weeds / Heinz Müller-Schaerer | 275 |
| Integrated Plant Control: System and Management / Ján Gallo | 279 |
| International Pesticide Poisoning Surveillance / Nida Besbelli and Jenny Pronczuk | 283 |

| | |
|--|-----|
| Invasive Insects as Major Pests in the United States / <i>E. Richard Hoebeke</i> | 288 |
| IPM Farmer Field School / <i>John Pontius</i> | 292 |
| Irradiation / <i>Hitoshi Ito</i> | 295 |
| Landscape Ornamentals / <i>Michael J. Raupp and Paula M. Shrewsbury</i> | 298 |
| Lawn-Care Treatments: Weeds / <i>Harlene Hatterman-Valenti</i> | 303 |
| Legal Aspects of Pesticide Applications / <i>Maristella Rubbiani</i> | 307 |
| Less Hazardous Alternatives: Promotions / <i>Luis Brenes and Luis Felipe Arauz Cavallini</i> | 310 |
| Lettuce Diseases: Ecology and Control / <i>Krishna V. Subbarao and Steven T. Koike</i> | 313 |
| Locust Control by Early Identification of Breeding Sites / <i>Arnold van Huis</i> | 319 |
| Lygus Bug Management by Alfalfa Harvest Manipulation / <i>Charles G. Summers, Shannon C. Mueller and Peter B. Goodell</i> | 322 |
| Mammal Trapping / <i>Gilbert Proulx</i> | 326 |
| Mammalia Pest Impacts in New Zealand / <i>Phil Cowan</i> | 329 |
| Mass-Trapping / <i>Masashi Kakizaki</i> | 332 |
| Mating Disruption / <i>Ring Carde</i> | 336 |
| Mechanical Weed Control in Agriculture / <i>Charles L. Mohler</i> | 338 |
| Mechanisms of Resistance to Agrochemicals / <i>Derek Hollomon</i> | 344 |
| Mitigating Impacts of Terrestrial Invasive Species / <i>Kathleen Fagerstone</i> | 347 |
| Mosquitoes: Biology / <i>Daniel L. Kline</i> | 350 |
| Mosquitoes: Control / <i>Daniel L. Kline</i> | 353 |
| Mosquitoes: Human Attacks / <i>Eric J. Hoffman, Edward D. Walker and James R. Miller</i> | 356 |
| Mulches and Pests / <i>Lars Olav Brandsæter</i> | 360 |
| Multilateral Environmental Agreements / <i>Ulrich Schlottmann and Mirco Kreibich</i> | 363 |
| National Pesticide Poisoning Surveillance / <i>Hans Persson and Margareta Palmborg</i> | 367 |
| Natural Enemies and Biocontrol: Artificial Diets for Rearing / <i>Simon Grenier and Patrick De Clerq</i> | 370 |
| Natural Enemies and Biocontrol: Function in Mixed Cropping Systems / <i>Tibor Bukovinszky, Joop C. van Lenteren and L.E.M. Vet</i> | 373 |
| Natural Enemies and Biocontrol: Monitoring / <i>Thomas W. Culliney</i> | 377 |
| Natural Enemies and Biocontrol: Quality Control Guidelines and Testing Methods / <i>Norman C. Leppla and Barbra C. Larson</i> | 382 |
| Natural Enemies: Destruction by Pesticides / <i>Joseph D. Cornell</i> | 385 |
| Natural Vegetation Management to Improve Parasitoids in Farming Systems / <i>Giovanni Burgio, Alberto Lanzoni and Antonio Masetti</i> | 387 |
| Nematicides / <i>Stephen R. Koenning</i> | 392 |
| Neurological Effects of Insecticides / <i>Michael E. Scharf</i> | 395 |
| Non-Indigenous Species: Crops and Livestock / <i>David Pimentel and Anne Wilson</i> | 400 |
| Non-Indigenous Species: Pests / <i>J. Howard Frank</i> | 404 |
| No-Till and Pest Problems / <i>Punnee Soonthornpoc</i> | 407 |
| Nozzle Types / <i>Alvin R. Womac</i> | 410 |
| Obsolete Pesticides: Management / <i>Alemayehu Wodageneh</i> | 412 |
| Oils / <i>Deepak Raj Khajuria and Divender Gupta</i> | 416 |
| Olive Insects: Ecology and Control / <i>Marshall W. Johnson and Kent M. Daane</i> | 425 |
| Organic Soil Amendments / <i>Philip Oduor Owino</i> | 428 |
| Ornamental Crop Pest Management: Plant Pathogens / <i>D. Michael Benson</i> | 432 |
| Papaya Diseases: Ecology and Control / <i>Arevik Poghosyan, Gina Holguin Zehfuss and Macario Bacilio Jimenez</i> | 435 |
| Papaya Insects: Ecology and Control / <i>Alberto Pantoja and Jorge E. Peña</i> | 440 |
| Parasites on <i>Oulema (Lema) lichenis</i> Voet, 1826 / <i>Ján Gallo</i> | 446 |

| | |
|---|-----|
| Pathogen-Free Stock: Managing Viruses and Viroids / Rudra Singh | 449 |
| Pathogens of Mass Produced Natural Enemies for Biocontrol / Susan Bjornson | 453 |
| Pea Diseases: Ecology and Control / Bruce D. Gossen and Sheau-Fang Hwang | 457 |
| Pea Insects: Ecology and Control / Juliana J. Soroka and Héctor A. Cárcamo | 461 |
| Peach Diseases: Ecology and Control / Alan R. Biggs | 465 |
| Peanut Diseases: Ecology and Control / Barbara B. Shew and Farid Waliyar | 469 |
| Pear Insects: Ecology and Control / David R. Horton and Thomas R. Unruh | 474 |
| Pecan Insects: Ecology and Control / Marvin K. Harris | 478 |
| Persistent Organic Pesticides / Agneta Sundén Byléhn | 481 |
| Pest Eradication: Screwworm as a Model / Robert E. Reichard | 484 |
| Pest Tolerance in Crops / Vicki Tolmay | 487 |
| Pest-Free Planting Stock: Selection / V. J. Shivankar and Shyam Singh | 490 |
| Pesticide Labels / William Smith | 495 |
| Pesticide Reduction: Strategies / Douglas L. Murray and Peter L. Taylor | 499 |
| Pheromone Traps and Trapping / Jenn-Sheng Hwang and Chau-Chin Hung | 502 |
| Pheromone Use in Integrated Pest Management of Stored Products / Pasquale Trematerra | 507 |
| Physical Barriers: Vertebrate Pests / Carolyn J. Randall | 511 |
| Phytosanitary Quarantine as a Pest Control Method / John S. Hartung | 516 |
| Plant Breeding / Gurdev S. Khush | 519 |
| Plant Food to Enhance Performance of Natural Enemies in Mass Rearing and the Field / Felix L. Wäckers and P.C.J. van Rijn | 524 |
| Poison Baits / Gilbert Proulx | 527 |
| Poisonous Arthropods / Findlay E. Russell | 531 |
| Pollution of the Environment / Joseph D. Cornell | 538 |
| Prescriptive Use of Pesticides / Harold Coble | 542 |
| Protected Crops Pest Management: Insects and Mites / Ramon Albajes | 544 |
| Quality Control of Formulations / Árpád Ambrus and László Bura | 548 |
| Reducing Pesticide Use: Successes / David Pimentel and Maria V. Cilveti | 551 |
| Resistance to <i>Bt</i> Transgenic Plants / Hugo Cerda-Perez and Denis J. Wright | 553 |
| Rice Diseases: Ecology and Control / T. S. Thind and Vineet K. Sharma | 556 |
| Rodent Exclusion / Krishnoji Rao Muktha Bai | 562 |
| Rodenticides / Alan MacNicoll | 567 |
| Rotterdam Convention and Pesticides / Barbara Dinham | 570 |
| Rouging / Hector Achicanoy | 573 |
| Runoff / Livia Nemeth Konda | 575 |
| Safe Use of Pesticides: A Developing Country's Point of View / Catharina Wesseling, Clemens Ruepert and Fabio Chaverri | 581 |
| Safe Use: Farmers' Association Point of View / Alarik Sandrup | 585 |
| Safe Use: Industry Point of View / John L. Aston | 588 |
| Safe Use: Regulator's Point of View / Nguyen Huu Huan | 591 |
| Satellite Imagery in Pest Management / John LeBoeuf | 594 |
| Secondary Pest Resurgence / C. N. Rao, V. J. Shivankar and Shyam Singh | 597 |
| Sheep Predation / Eugene L. Fytche | 602 |
| Silverleaf Whitefly Management Using Reflective Plastic and Wheat Straw Mulch / Charles G. Summers, James J. Stapleton and Jeffrey P. Mitchell | 606 |
| Soil Infiltration by Pesticides / George Antonious | 610 |
| Soil Management Practices / Randall C. Reeder | 614 |
| Sorghum Insects: Ecology and Control / Gerald E. Wilde | 618 |

| | |
|---|-----|
| Soybean Diseases: Ecology and Control / <i>Glen L. Hartman</i> | 623 |
| Strategies for Reducing Risks with Agricultural Pesticides in Developing Countries / <i>Sylvia I. Karlsson</i> | 626 |
| Strawberry Arthropods: Ecology and Control / <i>Noubar J. Bostanian and Joseph Kovach</i> | 630 |
| Strawberry Insects and Mites in California: Ecology and Control / <i>Frank G. Zalom, Douglas V. Shaw and Kirk D. Larson</i> | 634 |
| Submerged Aquatic Weeds: Costs and Benefits of Mechanical and Chemical Control / <i>Rohan D.S. Wells and John S. Clayton</i> | 637 |
| Sugarcane Diseases: Ecology and Control / <i>Stuart Rutherford</i> | 643 |
| Sunflower Diseases: Ecology and Control / <i>Tom Gulya</i> | 647 |
| Surveillance / <i>David J. Horn</i> | 652 |
| Sweetpotato Diseases: Ecology and Control / <i>Christopher A. Clark</i> | 655 |
| Synergy with Microorganisms / <i>Albrecht M. Koppenhöfer</i> | 658 |
| Systematics and Biological Pest Control / <i>Amy Y. Rossman and Michael Schauff</i> | 661 |
| Systemic Insecticides / <i>Nick C. Toscano and Nilima Prabhaker</i> | 664 |
| Tea Diseases: Ecology and Control / <i>N. Muraleedharan and U.I. Baby</i> | 668 |
| Tea Insects: Ecology and Control / <i>N. Muraleedharan</i> | 672 |
| Temperate-Climate Fruit Crop Pest Management: Plant Pathogens / <i>David F. Ritchie</i> | 675 |
| Tillage and Cultivation / <i>Joseph Ingerson-Mahar</i> | 680 |
| Unisexual Parasitoids in Biological Control / <i>Richard Stouthamer</i> | 683 |
| Vegetable Crop Pest Management: Insects and Mites / <i>Mark G. Wright and Mike Hoffmann</i> | 686 |
| Virus for the Biological Control of Insects / <i>Marlinda Lobo de Souza, Maria Elita Batista de Castro and Flavio Moscardi</i> | 689 |
| Vole Management for Orchards / <i>David Lockwood</i> | 693 |
| Weed Management: Biological and Chemical Approaches / <i>Thomas W. Culliney</i> | 696 |
| Weed Management: Home Landscaping / <i>Stephen C. Weller</i> | 699 |
| Weed Management: Introduction and Mechanical and Cultural Approaches / <i>Thomas W. Culliney</i> | 702 |
| Weed Management: Ornamental Nurseries / <i>Stephen C. Weller</i> | 705 |
| Weed Seed Dormancy: Implications for Weed Management Strategies / <i>Diego Batlla and Roberto Benech-Arnold</i> | 708 |
| Weeds and Carbon Dioxide / <i>Lewis H. Ziska</i> | 712 |
| West Nile Virus and Mosquito Control / <i>David Pimentel</i> | 715 |
| Wood Preservation / <i>H. Michael Barnes</i> | 719 |
| Worker Pesticide Exposure / <i>Myna Panemangalore, Avinash M. Tope and Frederick N. Bebe</i> | 722 |
| Worker Protection Standard / <i>James F. Ellerhoff and Joyce S. Hornstein</i> | 725 |
| Index | I-1 |

Preface

The Encyclopedia of *Pest Management* focuses on the identification and management of diverse pest species that damage and/or destroy food and livestock products as well as home and forest products. Throughout the world we are faced with rapid growth in the human population. Insuring that all our food and other needs are met is a prime concern for everyone.

Of major importance is the growing threat to the security of the human food supply. Signaling the seriousness of the human population explosion to our food security is the recent World Health Organization report that indicates that, currently, more than 3.7 billion people are malnourished. This is the largest number and proportion of malnourished ever reported in history. Malnourishment is a serious disease itself but it also increases human susceptibility to other debilitating diseases like malaria, diarrhea, and AIDS. Sick and diseased people find it difficult to work and even enjoy the other daily activities of their lives.

More stringent efforts are needed to conserve and protect the basic environmental resources that sustain the food system. These resources include fertile land, water, and energy, as well as diverse biological resources. Consider that more than 99.7% of the world's food is produced on the terrestrial ecosystem, while less than 0.3% of the food is produced in the oceans and other aquatic ecosystems. Looking to the future, more food will have to come from the land and less from the oceans. Urgently needed are safe, successful control methods for the destructive pests that ravage and destroy food and the other resources that sustain a productive agricultural system.

Worldwide, more than 40% of world food production is lost because crops are destroyed by insects, diseases, weeds, and some vertebrate animals. This tremendous loss is occurring despite the application of about 3 billion kilograms of pesticides and other pest controls now being used in world agriculture. Once the crops are harvested, other insect, microbe, and vertebrate pests destroy an additional 25% during storage and transport. As a result, more than half of all food produced is lost to pests, despite efforts to protect it. Clearly, everything possible must be done, both preharvest and postharvest, to reduce the loss of food to pests. Renewed efforts to more effectively protect food crops, as well as livestock, home and forest products, must become high priority.

The scientific leaders in pest management throughout the world have contributed to this encyclopedia. All articles were peer reviewed to reinforce their accuracy and objectivity. The articles assess the benefits and risks of various pest-management technologies. The use of pesticides as well as non-chemical controls are included, with every effort made to use quantitative data. In addition, they discuss the environmental and public-health impacts of pest control. We anticipate frequent updates as new information on pest management becomes available.

The editor is grateful to the specialists and colleagues throughout the world who are experts in the field of pest management and control. They have provided valuable advice and assistance concerning specific pests and management practices. In addition, the Advisory Board members, Susan Lee, Encyclopedia Editor and Supervisor, and Anne Wilson, my Research Assistant, gave tremendous support, guidance, and assistance in the development and production of this volume.

David Pimentel
Editor

About the Editor

David Pimentel is Professor of Insect Ecology and Agricultural Sciences, Department of Entomology and Department of Ecology and Evolutionary Biology, Cornell University, Ithaca, New York. The author or coauthor of over 600 scientific publications including 24 books, Dr. Pimentel has served or is serving with the National Geographic Society Research Committee, the Climate Institute, the International Food Policy Institute, the Royal Swedish Academy of Sciences, and the Chinese Academy of Science, among others. He received the B.S. degree (1948) from the University of Massachusetts, Amherst, and the Ph.D. degree (1951) from Cornell University, Ithaca, New York.

Encyclopedia of

Pest Management

Volume II

Adj-Cii

Coco-Field

Fil-Ins

Int-Mosq

Mulch-Path

Pea-Qual

Red-Sub

Sug-Work

Adjuvants and Carriers

Nan Lin

Vincent F. Garry

Department of Laboratory Medicine and Pathology, University of Minnesota, Minneapolis, Minnesota, U.S.A.

INTRODUCTION

Agricultural adjuvants are chemicals added to pesticides and pesticide mixtures in order to aid the operation and improve the effectiveness of the active ingredients. These agents are ubiquitous in terms of application, formulation, and chemical composition. Adjuvants greatly improve the efficiency and target-specificity of pesticides, and therefore play an important role in modern agriculture. In general, adjuvants do not pose a significant acute toxic hazard. However, recent studies suggest that adjuvants can complicate the toxicity of commercial-grade pesticides either by additive or independent effects of the active ingredients, and therefore further toxicological studies and enhanced regulatory oversight are required.

DEFINITION OF ADJUVANTS AND CARRIERS

Adjuvants

Agricultural adjuvants are chemicals added to pesticides and pesticide mixtures in order to aid the operation and improve the effectiveness of the active ingredients. At the beginning of the 20th century, animal proteins such as calcium caseinate were used to improve the performance of pesticides. Today, adjuvants as a major product group contain hundreds of chemicals with properties of wetting agents, spreaders, emulsifiers, thickening agents, dispersing agents, foaming adjuvants, foam suppressants, and penetrants.^[1] Adjuvants are used as components in commercial pesticide formulations, or mixed with pesticides before application as separate components, in order to enhance the performance of the active ingredients. Adjuvants can constitute as much as 60% of the volume of commercial-grade agrochemicals.^[2] In general, as part of a pesticide application mix, adjuvants may be applied at 0.25–1% (v/v) dilution, or at a rate of up to 1 gal/acre with spray solution.^[3,4] As a general rule, adjuvants are multi-component (complex) chemical mixtures. For example, polyoxyethylenes, polyvinyl compounds, and paraffin oils are often used together as adjuvants in one commercial pesticide product.^[3,5]

Carriers

Carriers serve as vehicles to ensure uniform distribution of formulated pesticides upon application.^[6] Liquid carriers include water and, in some cases, vegetable oils; solid carriers include pellets and granular materials such as attapulgite (a purified hydrated aluminum magnesium silicate) and kaolinite (clay minerals).^[6] Solid carriers are also used for stabilization of the active ingredients and add convenience to shipping and handling.^[7] Carriers do not necessarily improve the performance of the active ingredients as do adjuvants.

MODE OF ACTION OF ADJUVANTS

Adjuvants improve the performance of pesticides by enhancing penetration, improving solubility, or increasing retention of one or more active ingredients.^[3,8] Adjuvants can be divided into two broad categories by mode of action: 1) “spray modifiers” that alter the wetting, spreading, or sticking of the spray complex and 2) “activator adjuvants” that directly interact with the plant cuticle and increase absorption.^[9] Occasionally, anti-foam agents, buffering agents, and compatibility agents are listed as a third category, “utility adjuvants.”^[10]

Broadly classed, surfactants contribute to the majority of adjuvants. Traditional surfactant-type adjuvants cover a broad spectrum of nonionic, cationic, and anionic surfactants. An emerging class of surfactants, organosilicone surfactants such as polyether- and alkyl-silicones, are also widely used as adjuvants. Organosilicones demonstrate outstanding spreading abilities.^[11–13] By reducing surface tension, surfactant adjuvants increase spray droplet spread and work as “spray modifiers.” On the other hand, surfactant adjuvants increase retention and penetration through plant cuticle, and work as “activator adjuvants.” Additionally, surfactants also increase the pesticide absorption and shorten the acceptable time between application and rainfall (rainfastness).^[14] In some cases, surfactant adjuvants modify the mobility of active ingredients in soil.

Adjuvants such as Monazoline-O (oleyl imidazoline) and E-17-2 (dihydroxyethylisotridecyloxy-propylamine) reduce the mobility of the active ingredients of Norflurazon in soil. This property may reduce herbicide

leaching into groundwater and increase the efficiency of weed control.^[15]

The mode of action of nonsurfactant adjuvants is less well studied. Nitrogen fertilizers such as ammonium sulfate are applied as an adjuvant component in the herbicide spray solution.^[9] Ammonium salts improve the performance of herbicides, particularly weak acid herbicides. The mechanism remains unclear. Oils, including paraffin oils and vegetable oils, are another class of adjuvants. These short chain aliphatic hydrocarbons increase the penetration of active ingredients to the plant cuticle and solubilize less water-soluble active ingredients.^[6] In crop oils and crop-oil concentrates, up to 15–20% (v/v) non-ionic surfactants are mixed with oils upon application in order to combine the penetration property of oils and surface tension reducing property of the surfactants.^[14]

TOXICOLOGY OF ADJUVANTS AND RECENT RESEARCH ADVANCES

Most surfactant and oil adjuvants are reported to have nonspecific toxicity to mammals at high concentrations. Eye, dermal, and respiratory irritation is observed in animals and humans upon high levels of exposure.^[16] Paraffin oils, such as C14–17-chlorinated paraffins, induced biochemical and histological changes at 500–5000 ppm in rat liver and kidney in sub-chronic studies.^[17] Paraffin oils also cause irritation to the respiratory tract and skin in humans and animals.^[16]

In toxicity evaluation, it is common practice to examine each of the chemicals in an adjuvant formulation individually for their acute and chronic animal toxicity. However, in usage, adjuvants are complex chemical mixtures. Given the meager data available regarding the toxicity of these complex mixtures, the relative toxicity of adjuvants and other “inert” ingredients in pesticide formulations has been and remains understudied. Recent research demonstrated that adjuvants can complicate the toxicity of commercial-grade pesticides and pesticide mixtures by producing adverse effects independent of the active ingredients or by enhancing the toxicity of the active ingredients.^[18]

One of the most common mechanisms of surfactant toxicity is the disruption of cell membrane integrity. Surfactant-induced phytotoxicity (plant cell membrane toxicity) has been detected by measuring ethylene evolution and pigment efflux.^[19,20] These effects may lead to inhibition of growth and development, inhibition of seed germination, and necrosis of leaf tissues.^[20] Viewed on a molecular level, surfactants interact with membrane phospholipids and proteins, increase permeability, and eventually cause leakage of low molecular weight cytosolic compounds, cell damage, and cell death.^[18] Surfactant-enzyme binding can alter enzyme activity

(Cserhati, *ibid*). For example, surfactant Emulgen 913 (polyoxyethylene glycol nonylphenyl ether) was reported to decrease liver P450 and cytochrome *b*₅ content, and to increase heme oxygenase activity in rats.^[21,22]

Some adjuvants exert genotoxicity *in vitro*. X-77 (a combination of alkyl polyoxyethylenes, free fatty acids, glycol and isopropanol), Nacol-trol [polyvinyl polymer (polyarylamide) and inert ingredients], Preference (a proprietary surfactant containing less than 0.002% ethylene oxide and less than 0.002% dioxane), and Direct (polyvinyl polymers and inert ingredients) gave dose-dependent increased frequency of micronuclei *in vitro*.^[8] Oxyethylene adjuvants can contain impurities such as ethylene oxide, a known human mutagen.^[23,24]

More recently, some reagent-grade alkylphenols (APs) and alkylphenol ethoxylates (APEs)-derived adjuvants were found to act as endocrine disruptors in various *in vitro* and *in vivo* assays.^[25] Further, commercial-grade X-77 and Activate Plus, two alkylphenol ethoxylate adjuvants, were found to stimulate MCF-7 human breast cancer cell proliferation.^[2] It has been proposed that long-chain APs and APEs could be biotransformed into short-chain derivatives, such as nonyl- and octylphenols, and exert estrogenicity.^[26,27] Upon metabolism of surfactant octylphenol polyethoxylate (OPP) to octylphenol (OP), male rats were observed to have significant reduction in mean testicular size upon prenatal oral exposure to OP and OPP at a concentration of 1000 µg/L in drinking water.^[26]

REGULATORY ISSUES

“Good adjuvants make ordinary chemicals do extraordinary things.”^[28] Adjuvant selection is a critical, if not the most important, factor in a competitive pesticide product.

They are often labeled as “inert ingredients” in product labeling and material safety data sheets (MSDSs). As such, adjuvant content and composition may go unrecognized in these pesticide products. Regarding the composition of commercially available adjuvant formulations, non-proprietary information regarding these ingredients is routinely provided to users; proprietary information may be available in case of emergency.^[7]

Active ingredients undergo mandatory testing and registration processes at regulatory agencies, while registration of adjuvants is largely overlooked. However, some states such as California, Washington, and Idaho do require registration of adjuvants; numerous states do not have adequate oversight regarding adjuvant application.^[14] Potential adverse effects of adjuvants have drawn the attention of regulatory agencies such as the Environmental Protection Agency (EPA) in risk assessment of pesticide application.^[28] Increased research effort regarding adjuvant toxicology is expected.

ACKNOWLEDGMENTS

The authors thank Ms. C. Ehlen for assistance in editing this manuscript.

REFERENCES

1. Meister. *Farm Chemicals Handbook '97*; Meister Publishing Company, 1997; C10.
2. Lin, N.; Garry, V.F. In vitro studies of cellular and molecular developmental toxicity of adjuvants, herbicides, and fungicides commonly used in red river valley, Minnesota. *J. Toxicol. Environ. Health* **2000**, *60* (6), 423–439.
3. Harvey, L.T.E. *A Guide to Agricultural Spray Adjuvants Used in the United States*; Thompson Publications: Fresno, California, 1992; 1–195.
4. Witt, W.W. Adjuvants. **2001**, <http://www.ca.uky.edu/agripedia/pls404/adjuvant.htm>.
5. Schonherr, J.A.; Bauer, H. Adjuvants for agrochemicals. In *Analysis of Effects on Surfactants on Permeability of Plant Cultures*; CRC Press: Boca Raton, Florida, 1992; 17–35.
6. McCarty, L.B. Spray additives and pesticide formulations. In *Florida Lawn Handbook: An Environmental Approach to Care and Maintenance of Your Lawn*, 1st Ed.; University Press of Florida: Gainesville, Florida, 1995; SP-45, <http://edis.ifas.ufl.edu/LH061>.
7. Morgan, D.P.E. Miscellaneous pesticides, solvents, and adjuvants. In *Recognition and Management of Pesticide Poisonings*, 4th Ed.; U.S. Environmental Protection Agency: Washington, DC, 1989; 146–158, <http://www.epa.gov/ncepihom/nepishom/tips.html>.
8. Garry, V.F.; Burroughs, B.; Tarone, R.; Kesner, J.S. Herbicides and adjuvants: an evolving view. *Toxicol. Ind. Health* **1999**, *15* (1–2), 159–167.
9. Curran, W.S.; McGlamery, M.D.; Liebl, R.A.; Lingenfelter, D.D. *Agronomy Facts 37: Adjuvants for Enhancing Herbicide Performance*; Pennsylvania State University: University Park, Pennsylvania, 1999; <http://pubs.cas.psu.edu/FreePubs/uc106.html>.
10. Witt, J.M. *Spray Adjuvant Fact Sheet*; The Pesticide Management Education Program, Cornell University: Ithaca, New York, 1998; <http://pmep.cce.cornell.edu/facts-slides-self/facts/gen-peapp-adjuvants.html>.
11. Roggenbuck, F.C.; Rowe, L.; Penner, D.; Petroff, L.; Burow, R. Increasing postemergence herbicide efficacy and rainfastness with silicone adjuvants. *Weed Technol.* **1990**, *4* (3), 576–580.
12. Roggenbuck, F.C.; Penner, D.; Burow, R.F.; Thomas, B. Study of the enhancement of herbicide activity and rainfastness by an organosilicone adjuvant utilizing radiolabelled herbicide and adjuvant. 3rd Int. Symp. Adjuv. Agrochem. Cambridge, England, UK, **1992**, *37* (2), 121–125.
13. Stevens, P.J.; Kimberley, M.O.; Murphy, D.S.; Policello, G.A. Adhesion of spray droplets to foliage the role of dynamic surface tension and advantages of organosilicone surfactants. 3rd Int. Symp. Adjuv. Agrochem. Held by The Physicochemical and Biophysical Panel of the Sci. **1992**, *38* (2–3), 237–245.
14. Witt, W.W. Adjuvants. In *Agripedia*; University of Kentucky: Lexington, Kentucky, 2001; <http://www.ca.uky.edu/agripedia/pls404/adjuvant.htm>.
15. Chandran, R.S.; Singh, M. Reduction of norflurazon leaching in a sandy soil by adjuvants. *Bull. Environ. Contam. Toxicol.* **1999**, *62* (3), 315–323.
16. Reigart, J.R.; Roberts, J.R. *Recognition and Management of Pesticide Poisonings*, 5th Ed.; Office of Pesticide Programs, U.S. Environmental Protection Agency: Washington, DC, 1999; 18, Section V, <http://www.epa.gov/pesticides/safety/healthcare/handbook/contents.htm>.
17. Poon, R.; Lecavalier, P.; Chan, P.; Viau, C.; Hakansson, H.; Chu, I.; Valli, V.E. Subchronic toxicity of a medium-chain chlorinated paraffin in the rat. *J. Appl. Toxicol.* **1995**, *15* (6), 455–463.
18. Cserhati, T. Alkyl ethoxylated and alylphenol ethoxylated nonionic surfactants: interaction with bioactive compounds and biological effects. *Environ. Health Perspect.* **1995**, *103* (4), 358–364.
19. Coupland, D.; Zabkiewicz, J.A.; Ede, F.J. Evaluation of three techniques used to determine surfactant phytotoxicity. *Ann. Appl. Biol.* **1989**, *115* (1), 147–156.
20. Matsui, H.; Shafer, W.E.; Bukovac, M.J. Surfactant-induced ethylene evolution and pigment efflux from beet (*beta vulgaris l.*) root tissue. In *Adjuvants for Agrichemicals*; CRC Press: Boca Raton, Florida, 1992; 59–76.
21. Ariyoshi, T.; Shiiba, S.; Hasegawa, H.; Arizono, K. Profile of metal-binding proteins and heme oxygenase in red carp treated with heavy metals, pesticides and surfactants. *Bull. Environ. Contam. Toxicol.* **1990**, *44* (4), 643–649.
22. Ariyoshi, T.; Hasegawa, H.; Matsumoto, H.; Arizono, K. Effects of surfactants on the contents of metallothionein, heme and hemoproteins and on the activities of heme oxygenase and drug-metabolizing enzymes in rats pretreated with phenobarbital or betanaphthoflavone. *Bull. Environ. Contam. Toxicol.* **1991**, *46* (1), 120–127.
23. Garry, V.F.; Hozier, J.; Jacobs, D.; Wade, R.L.; Gray, D.G. Ethylene oxide: evidence of human chromosomal effects. *Environ. Mutagen.* **1979**, *1* (4), 375–382.
24. Landrigan, P.J. Environmental pollution and health. *Lancet* **1992**, *340* (8829), 1220.
25. Nimrod, A.C.; Benson, W.H. Environmental estrogenic effects of alkylphenol ethoxylates. *Crit. Rev. Toxicol.* **1996**, *26* (3), 335–364.
26. Sharpe, R.M.; Fisher, J.S.; Millar, M.M.; Jobling, S.; Sumpter, J.P. Gestational and lactational exposure of rats to xenoestrogens results in reduced testicular size and sperm production. *Environ. Health Perspect.* **1995**, *103* (12), 1136–1143.
27. Soto, A.M.; Sonnenschein, C.; Chung, K.L.; Fernandez, M.F.; Olea, N.; Serrano, F.O. The E-screen assay as a tool to identify estrogens: an update on estrogenic environmental pollutants. *Environ. Health Perspect.* **1995**, *103* (Suppl. 7), 113–122.
28. McCarthy, J.F. Regulatory issues and adjuvants. In *Adjuvants for Agrichemicals*; CRC Press: Boca Raton, Florida, 1992; 245–246.

Aerial Ultra-Low-Volume Application of Insecticide to Control Adult Mosquitoes: Minimizing Non-Target Impacts

He Zhong

Public Health Entomology Research and Education Center, Florida A&M University, Panama City, Florida, U.S.A.

INTRODUCTION

Aerial ultra low volume (ULV) application of mosquito adulticides is one of the most effective techniques for controlling mosquitoes and preventing mosquito-borne diseases.^[1] During application, large insecticide droplets may sometimes be deposited onto tidal wetlands or wildlife habitat that can result in unwanted mortality to non-target organisms.^[2,3,4-8] For many years, when controlling mosquitoes, non-target mortality caused by an adulticiding operation was usually accepted as a ‘casualty of war.’ With the advancement of new spray technologies, we now recognize that these ‘casualties of war’ can be reduced to a minimum. The important factors that contribute to mosquito control efficacy and non-target mortality are insecticide deposition,^[7-9] droplet size,^[1,10] spray time,^[10] application dose,^[1,10] topography,^[10] and weather conditions, such as wind velocity, direction, temperature, and atmospheric stability.^[10] Due to the complexity of aerial ULV applications, it may be difficult to achieve an ideal level of adult mosquito control without non-target mortality during a spray mission. But control efficacy can be increased, and non-target organism impact minimized, if aerial application is conducted at the right place (by increasing retention time of mosquitoicide droplets in the air in order to enhance their contact with flying mosquitoes),^[11,12] at the right time (dusk, dawn, or night when adult mosquitoes are actively flying),^[10] and at the right dose (proper application rate to kill mosquitoes but not non-target organisms).^[1,9]

RIGHT PLACE

Following ULV application, insecticide droplets need to remain airborne for effective control of adult mosquitoes. The size of droplets governs downwind dispersal and subsequent impingement on targets.^[1] The probability of droplet impingement onto flying mosquitoes is increased if droplet retention time in the

air is increased. Smaller droplets [5–25 µm volume median diameters (VMD)] are retained in the air column longer and for greater impingement on adult mosquitoes.^[1] Larger insecticide droplets (>100 µm VMD) will deposit on the ground more quickly after application, thereby reducing the likelihood of contact with flying mosquitoes. Insecticide that is deposited on the ground is not only wasted, but may adversely affect non-target organisms.^[7,8] Environmental contamination can be reduced by adopting application techniques that maintain droplets in the air, promote controlled downwind movement of the insecticide cloud, while minimizing ground deposition (particularly in environmentally-sensitive areas).^[7,9] This concept is different from agricultural applications, where deposition is needed to coat the surface of crops with insecticides. Moreover, agricultural applications try to reduce insecticide drift off the target zone, such as a crop or field, rather than maximize droplet suspension in the air column.

RIGHT TIME

The best time of the day for adulticide applications (also called the ‘spray window’) is at dusk, dawn, or nighttime.^[10] Applying an insecticide at that period will reduce impact on daytime active non-target organisms such as honeybees, dragonflies, and butterflies. These are the time periods when daytime non-targets are resting and, therefore, protected from insecticide exposure. Maximum adult mosquito control can be achieved using this spray window because most mosquitoes are actively flying during this time.^[1] Currently, many mosquito control programs have adopted such spray windows. This practice is a significant achievement that protects many daytime active non-targets from being exposed to the insecticide spray cloud.

Spraying at the right time also means spraying under optimal meteorological conditions. Understanding, as well as achieving, ‘optimal’ meteorological conditions for mosquito spraying is often difficult.^[1] Current aerial

mosquito application technology can accurately calibrate the amount of insecticide output from nozzle systems. However, after the insecticide is released from the spray system, the aerosol is in the hands of Mother Nature. The spray cloud, as it is carried by wind and influenced by gravity, starts its journey to the ground from an altitude of 100–300 ft. Wind velocity, direction, temperature, and atmospheric stability greatly affect the distribution of the spray cloud.^[1] Also, downwind movement and deposition of insecticide residue can vary greatly from one spray mission to another.^[1,2] This situation creates considerable variation in control efficacy and can often influence whether the effects on non-targets are minimal or substantial.

RIGHT DOSE

Increasing the application rate may increase the risk of non-target mortality due to escalating exposure levels. In reality, it is sometimes very difficult to apply the proper dosage to achieve adequate mosquito control without causing non-target mortality. Non-targets' differential tolerance to insecticides may be the result of physiological as well as geographical differences within and among those organisms. Also, natural topographic barriers, such as trees, bushes, and grasses can provide refuge for non-targets to escape exposure from the insecticide aerosol.^[8] However, mosquito control efficacy in vegetated areas may be reduced at the same time^[1] and if the application rate is increased to compensate for this, adverse effects on non-targets may occur.

To determine the proper application rate, studies need to be initiated in order to determine the relationship of the insecticide concentration in the air column with that of adult mosquito mortality^[11,12] and ground deposition of the insecticide with non-target mortality.^[7–9] The insecticide concentration in both target and non-target zones is called the terminal insecticide concentration (TIC), which is different from and can be influenced by application dose. The TIC is also influenced by many environmental variables and therefore needs to be frequently monitored. If the TIC is adequate to kill the majority of adult mosquitoes and low enough that it spares non-targets of concern, the application dose will be appropriate. In this way, the TIC critically affects control efficacy and non-target impact and it is necessary to incorporate the determination of a TIC into the routine application of insecticides. TIC data may also be used to assess or cross-compare control efficacy and impact on non-targets during aerial mosquito control missions. This process will ensure the proper application dose to achieve the delicate balance between effective mosquito control and minimal non-target impact.

NEW SPRAY TECHNOLOGY

Mosquito control programs worldwide are continuing to develop new spray technologies to promote better mosquito control efficacy and lessen damage to non-targets. In the late 1990s, James Robinson's group at Florida's Pasco County Mosquito Control District led an effort to develop a high-pressure nozzle system to deliver small insecticide droplets (<30 μm VMD).^[11,12] The high-pressure system, with two-third application rates, achieved better adult mosquito control compared to a conventional flat fan nozzle system.^[11] Zhong et al.^[7] compared insecticide residue deposition from these two aerial ULV spray systems in Collier County, Florida. Using fenthion as the test material, heavy ground deposits were found within one mile downwind of the application with the flat-fan nozzle system that resulted in 80% mortality of caged fiddler crabs, *Uca pugilator* (Bosc). On the other hand, minimal fenthion ground deposits were detected during the high-pressure nozzle trials. No fiddler crab mortality was observed within the 8-km downwind test area following three single-swath applications repeated during three consecutive nights.

The impact of naled on honeybees, *Apis mellifera* L., was investigated by exposing beehives to nighttime aerial ULV applications of insecticide naled using the flat-fan nozzle system,^[8] and later with the high-pressure nozzle system.^[9] The tests were conducted during routine aerial adult mosquito control missions in Manatee County, Florida. Honeybees, which clustered outside of hive entrances, were subjected to naled exposure during these spray missions. The flat-fan nozzle system killed over 90% of the honeybees clustered outside of the hives and resulted in an average of 35% reduction in honey yield at the end of the season.^[8] On the other hand, bee mortality from the high-pressure nozzle system was no greater than that of control hives not exposed to naled (5% on average). In that study, the average honey yield in treatment hives was not reduced when compared to control hives.^[9]

INSECTICIDE RESIDUE MONITORING

At present, bioassay techniques are widely used to measure mosquito control efficacy and non-target organism impact. Generally, bioassays only answer 'yes or no' (i.e., dead or alive) for most acute toxicity tests. Bioassays do not address critical insecticide residue issues such as 'Where or how much insecticide is present following the ULV application?' Chronic bioassays are often not conducted due to time and funding levels, etc. Based on the dose-response relationship, the TIC in the air column and data on

ground deposition is very important in controlling mosquitoes and causing impact on non-targets. When insecticide residue monitoring is used in conjunction with bioassays, mosquito control programs will have a powerful quantitative tool to combat mosquitoes while protecting the environment.^[11,12,7-9]

Insecticide residues cannot be observed by the human eye. However, they can be detected and quantified by modern analytical techniques such as gas chromatography or high performance liquid chromatography. By monitoring insecticide residues we can: (1) determine the actual concentration of insecticide in the air; (2) determine the actual concentration of insecticide deposited on the ground or into the water; (3) determine the distance of aerosol movement downwind; (4) establish appropriate thresholds for determining non-target impacts and mosquito control efficacy; and (5) compare different application equipment or operational scenarios.

CONCLUSION

In summary, controlling adult mosquitoes through aerial application of insecticides while minimizing non-target impacts can be achieved by operational mosquito control programs. Instituting a residue monitoring program can be helpful in protecting the environment. Additionally, a monitoring program can assist in determining the efficacy of insecticide applications as it relates to controlling mosquitoes so that the proper application of insecticides can be achieved at the right time, in the right place, and at the right dose.

ACKNOWLEDGMENTS

I thank James Cilek, Hyun-Woo Park, Susan Lee, and two anonymous reviewers for their critical reviews, helpful comments, and suggestions.

REFERENCES

1. Mount, G.A.; Biery, T.L.; Haile, D.G. A review of ultra-low-volume aerial sprays of insecticide for mosquito control. *J. Am. Mosq. Control. Assoc.* **1996**, *12* (4), 601–618.
2. Hill, E.F.; Eliason, D.A.; Kilpatrick, J.W. Effects of ultra-low volume application of malathion in Hale County, Texas. *J. Med. Entomol.* **1971**, *8* (2), 173–179.
3. McKenney, C.L.; Shirley, M.A.; Pierce, R.H. *A Two-Year Research Project in the Rookery Bay National Estuarine Research Reserve to Evaluate Populational and Physiological Responses of Crabs from Various Habitats in Relation to the Fate of a Mosquito Control Pesticide*; Contract No. NC-ND2100-3-00042; 1997; 9 pp.
4. Pankiw, T.; Jay, S.C. Aerially applied ultra-low-volume malathion effects on caged honey bees (Hymenoptera: Apidae), caged mosquitoes (Diptera: Culicidae), and malathion residues. *J. Econ. Entomol.* **1992**, *85*, 687–691.
5. Pankiw, T.; Jay, S.C. Aerially applied ultra-low-volume malathion effects on colonies of honey bees (Hymenoptera: Apidae). *J. Econ. Entomol.* **1992**, *85*, 692–699.
6. Schoor, W.P.; Middaugh, D.P.; Marcovich, D.T. Effects of aerially applied fenthion on survival and reproduction of the panacea sand fiddler, *Uca panacea*, in laboratory habitats. *Arch. Environ. Contam. Toxicol.* **2000**, *38*, 327–333.
7. Zhong, H.; Dukes, J.; Greer, M.; Hester, P.; Shirley, M.; Anderson, B. Ground deposition impact of aerially applied fenthion on the fiddler crabs, *Uca pugilator*. *J. Am. Mosq. Control. Assoc.* **2003**, *19*, 47–52.
8. Zhong, H.; Latham, M.; Hester, P.G.; Frommer, R.L.; Brock, C. Impact of aerial applied ultra-low-volume insecticide naled with flat-fan nozzle system on honeybee survival and productivity. *Arch. Environ. Contam. Toxicol.* **2003**, *45*, 216–220.
9. Zhong, H.; Latham, M.; Payne, S.; Brock, C. Minimizing the impact of mosquito adulticide naled on honeybees *Apis mellifera* (Hymenoptera: Apidae): aerial ultra-low volume application using a high-pressure nozzle system. *J. Econ. Entomol.* **2004**, *97*, 1–7.
10. Mount, G.A. A critical review of ultralow-volume aerosols of insecticide applied with vehicle-mounted generators for adult mosquito control. *J. Am. Mosq. Control. Assoc.* **1998**, *14* (3), 305–334.
11. Dukes, J.; Zhong, H.; Greer, M.; Hester, P.; Hogan, D.; Barber, J.A.S. A comparison of two spray nozzle systems used to aerially apply the ultra-low-volume adulticide fenthion. *J. Am. Mosq. Control. Assoc.* **2004**, *20* (1), 27–35.
12. Dukes, J.; Zhong, H.; Greer, M.; Hester, P.; Hogan, D.; Barber, J.A.S. A comparison of two ultra-low-volume spray nozzle systems by using a multiple swath scenario for the aerial application of fenthion against adult mosquitoes. *J. Am. Mosq. Control. Assoc.* **2004**, *20* (1), 36–44.

Alfalfa Diseases: Ecology and Control

Landon H. Rhodes

Department of Plant Pathology, The Ohio State University, Columbus, Ohio, U.S.A.

R. Mark Sulc

Department of Horticulture and Crop Science, The Ohio State University, Columbus, Ohio, U.S.A.

INTRODUCTION

Diseases are among the most significant factors limiting the yield and quality of alfalfa (*Medicago sativa* L.), one of the most important forage legume species grown worldwide. Alfalfa diseases are caused by biotic (infectious) and abiotic (non-infectious) agents.^[1] The latter group includes problems caused primarily by environmental extremes (drought, low temperature, and low soil pH), by deficits or imbalances of essential nutrients, or by pollutants. This article focuses on the biotic (infectious) pathogens, which include fungi, bacteria, viruses, phytoplasmas, nematodes, and parasitic plants. For an infectious disease to occur, there must be a pathogen, a susceptible alfalfa plant host, and an environment conducive to disease development.

Alfalfa diseases can be divided into several categories based on the plant parts attacked, the type of damage done to the plant, and to some extent, the pathogen group involved (Table 1). Some diseases could easily be placed in two or more categories. For example, anthracnose consists of both stem blight and crown rot phases. In general, however, the categories represent distinct types of diseases and serve to simplify an array of over 100 individual diseases that are known to occur on alfalfa worldwide.

DISEASE DEVELOPMENT

Understanding disease development involves identification of the source of inoculum and its means of dissemination. Many alfalfa pathogens reside in plant residue from the current or previous crop. As the residue decomposes, infective propagules of the pathogen (spores, bacterial cells, etc.) are gradually released into the soil. Such “soil-borne” pathogens are present when the alfalfa crop is planted and may infect plant roots as they grow through the soil. Alternatively, these propagules may be carried to plant roots by soil insects such as the clover root curculio, or, if close to the soil surface, may be splashed to above-ground plant parts. Some pathogens survive in the soil in a dormant state,

e.g., *Sclerotinia trifoliorum*, and produce specialized fruiting structures capable of releasing spores into the air. Millions of air-borne spores can be produced by a single fruiting body only one quarter of an inch in diameter. Spores landing on susceptible alfalfa plants rapidly germinate and penetrate the leaf and stem tissues.

Alfalfa is a perennial crop, so many pathogens build up on the crown and lower stems (stubble), which remain after harvest. *Phoma medicaginis*, the cause of spring black stem, overwinters in the stubble where it produces spores adapted to water-splash dissemination. New spring growth is readily infected by spores splashed from the previous year’s stubble. Some pathogens are spread from plant to plant by harvesting equipment. This is especially true for the vascular wilt pathogens, where spores within the stem xylem tissue are exposed when stems cut during harvest. Subsequent cutting of healthy plants by the contaminated harvest equipment leads to the deposition of the spores directly on cut surfaces where they can germinate and grow into the exposed xylem. Most viruses are carried to alfalfa plants by homopteran insects, most notably aphids.^[2] Alfalfa mosaic virus (AMV), for example, is carried by at least 14 species of aphid. Because AMV has a wide range of host plants (over 400 wild and cultivated species), it may be brought into alfalfa fields by aphids that previously fed on infected crop plants or weeds in nearby fields.

EFFECTS OF DISEASES ON ALFALFA

Diseases reduce chances for successful stand establishment, limit yields, and hasten stand decline of alfalfa. Effects of disease on individual plants vary widely. Some diseases are lethal, while others only cause stunting or leaf loss. Many of the seedling diseases, vascular wilts, and crown and root rot diseases result in plant death, sometimes within a matter of days. In contrast, most foliar diseases and those caused by viruses rarely kill affected plants. Diseases in which a single pathogen

Table 1 Major types of diseases affecting alfalfa worldwide

| Disease type | Specific examples | Pathogens | Symptoms |
|--------------------------------|--|---|---|
| Seedling diseases | Damping-off Aphanomyces root rot | <i>Pythium</i> and <i>Phytophthora</i> spp. ^a <i>Rhizoctonia solani</i> <i>Aphanomyces euteiches</i> | Collapse and decay of seedling tissues, often occurring first in the hypocotyl region; root rot in older seedlings |
| Foliar diseases | Common leaf spot Lepto leaf spot Spring black stem and leaf spot Stemphylium leaf spot Summer black stem and leaf spot Downy mildew | <i>Pseudopeziza medicaginis</i> <i>Leptosphaerulina trifolii</i> <i>Phoma medicaginis</i> <i>Stemphylium</i> spp. <i>Cercospora medicaginis</i> <i>Peronospora trifoliorum</i> | Dark spots (lesions) on leaves and stems, often associated with yellowing (chlorosis) and death (necrosis) of colonized and surrounding tissues; premature loss of leaflets or entire leaves; symptoms most pronounced on lower portion of plant; entire plant stunted if infection severe; reproductive bodies of the pathogen often present in tissue |
| Stem blights | Rhizoctonia stem blight Anthracnose | <i>Rhizoctonia solani</i> <i>Colletotrichum trifolii</i> | Discrete (anthracnose) or diffuse (<i>Rhizoctonia</i>) areas of tan to brown tissue. Upper stem and leaves bleached and often wilted |
| Root and crown rots | Phytophthora root rot Sclerotinia crown and stem rot Fusarium crown and root rot Brown root rot Southern blight | <i>Phytophthora medicaginis</i> <i>Sclerotinia trifoliorum</i> and <i>S. sclerotiorum</i> <i>Fusarium</i> spp. <i>Phoma sclerotioides</i> <i>Sclerotium rolfsii</i> | A variety of symptoms depending on pathogen. Dark lesions on taproot (esp. <i>Phytophthora</i> ; brown root rot); decay of taproot with resulting loss of distal portion; loss of feeder roots; soft, “mushy” crown rot (<i>Sclerotinia</i> ; southern blight); red-brown discoloration, and dry decay (<i>Fusarium</i>) |
| Vascular wilts | Fusarium wilt Verticillium wilt Bacterial wilt | <i>Fusarium oxysporum</i> f. sp. <i>medicaginis</i> <i>Verticillium albo-atrum</i> <i>Clavibacter michiganensis</i> subsp. <i>insidiosus</i> | Yellowing of leaves and stems; shortened upper internodes with twisted leaflets (<i>Fusarium</i> and <i>Verticillium</i>); discolored vascular tissue in taproot; overall stunting; eventual death of entire plant |
| Virus and phytoplasma diseases | Alfalfa mosaic Witches’ broom | Alfalfa mosaic alfamovirus (AMV) Phytoplasma | Patches or streaks of yellow on leaves; malformed leaves; proliferation of adventitious shoots (Witches’ broom); stunting |
| Nematode diseases | Stem nematode Lesion nematode Root knot nematode | <i>Ditylenchus dipsaci</i> <i>Pratylenchus</i> spp. <i>Meloidogyne</i> spp. | A variety of symptoms depending on pathogen. Bleached foliage with distorted stems (stem nematode); dark lesions on feeder roots (lesion nematode); spindle-shaped galls on roots (root-knot nematode) |
| Parasitic plant diseases | Dodder | <i>Cuscuta</i> spp. | Plants entangled in yellow, leafless stems of dodder plant |
| Abiotic diseases | Nutrient deficiencies | Inadequate nutrient supply | Yellowing; stunting. Symptoms vary depending on nutrient |

^aSpecies.

is capable of killing plants have been referred to as “acute” diseases.^[3] Examples of acute diseases include *Fusarium* wilt, anthracnose, *Phytophthora* root rot, and *Sclerotinia* crown and stem rot. Diseases that are typically nonlethal, such as Lepto leaf spot, alfalfa mosaic virus, and root-knot nematode have been termed “chronic” diseases. While the individual pathogens causing these diseases may not be capable of killing

plants outright, collectively they impose a “cumulative stress load” on a plant that ultimately may result in death.^[3] A third category includes those problems known as “disease complexes.” For example, crown rot is a lethal disease thought to be the result of the combined action of several pathogens growing in the crown region of the plant.^[3] *Fusarium* spp., *Rhizoctonia solani*, *Colletotrichum* spp., *Phoma* spp., *Pythium* spp., and other

pathogenic fungi, as well as *Pseudomonas* and other bacteria can often be isolated from the rotted crowns of dead and dying plants. The relative importance and interactions of each of these pathogens is still unclear.

CONSIDERATIONS FOR DISEASE CONTROL

Sound crop production practices will reduce the chances of serious losses to alfalfa productivity owing to disease, because they help maintain a vigorous alfalfa stand. Although some disease-causing agents are more likely to attack healthy plants rather than stressed plants, in general, any practice that improves plant vigor is likely to reduce the chance of plants becoming diseased. More importantly, good growing conditions will allow surrounding uninfected plants to achieve their maximum potential and compensate for the loss in stand or productivity because of diseased plants.

Field Selection

Alfalfa fields should be well drained. Fields subject to temporary flooding should be avoided, as many seedling diseases and root and crown rot diseases are favored by wet soil conditions.

Crop Rotation

Alfalfa cultivation should not follow alfalfa or other forage legumes in a rotation, as pathogens that have built up reservoirs of inoculum on the previous crop may attack the new crop as soon as it is planted. Wherever possible, alfalfa cultivation should follow corn, small grain, or other grass crop in the rotation.

Soil pH and Fertility

The soil pH should be 6.5–7.0. Application of lime to acid soils should be done 6–12 months in advance of seeding the alfalfa crop. Maintaining proper pH through appropriate liming usually meets the crop needs for magnesium, calcium, as well as micronutrients. Adequate phosphorus and potassium fertilization is critical for alfalfa plants to maintain their resistance to disease, particularly to the crown-rotting organisms. On some soils, sulfur and boron fertilization may be needed.

Variety Selection

Development of disease-resistant alfalfa varieties has been a major focus of public institutions and private seed companies over the past four decades. Recent

commercial alfalfa varieties have high levels of genetic resistance to bacterial wilt, Fusarium wilt, Verticillium wilt, anthracnose, Phytophthora root rot, and Aphanomyces root rot. However, it should be noted that an alfalfa variety is actually a mixture of several genetic types; not all plants within the variety will carry resistance to a particular disease, even though the variety may be listed as resistant to that disease. Standard designations have been developed to allow characterization of the percent of plants within a variety that have genetic resistance to a particular disease,^[4] ranging from highly resistant to susceptible (Table 2). For example, a variety that is listed as having resistance to Phytophthora root rot may actually have only 35–50% of the plants with genetic resistance. Thus, it is possible to have significant losses, even in varieties classified as resistant. It should also be noted that for many serious diseases, resistant varieties are not available.

Chemical Control

Few chemicals are available to control alfalfa diseases. Seed treatment formulations of metalaxyl (Allegiance) or mefenoxam (Apron XL) are effective in controlling seedling diseases caused by *Pythium* and *Phytophthora*. In most cases, commercial seed is treated at a seed treatment facility prior to being sold. Kocide DF (cupric hydroxide) and other copper-based fungicides may be available for use against certain foliar diseases such as Lepto leaf spot and summer black stem.

Control of Weed and Insect Pests

Usually weeds invade areas where alfalfa is declining due to some other cause. Weeds by themselves seldom crowd out alfalfa on fertile soils under timely harvest management. In some cases, severe weed encroachment may reduce alfalfa vigor and increase the susceptibility to disease. Insects may seriously weaken alfalfa

Table 2 Standard designations for disease resistance in an alfalfa variety

| Disease resistance levels in an alfalfa variety | |
|---|--------------------|
| Resistance level | % Resistant plants |
| Highly resistant (HR) | >50 |
| Resistant (R) | 31–50 |
| Moderately resistant (MR) | 15–30 |
| Low resistant (LR) | 6–14 |
| Susceptible (S) | 0–5 |

(From Ref.^[4])

plants and frequently wound plant tissue, providing entry points for crown- and root-rotting organisms.

Timely Harvest

Most leaf spot diseases reduce yield and quality by causing leaf drop. Harvests made on schedule will reduce the amount of leaf loss and also minimize the buildup of the disease for the subsequent cutting. Proper harvest schedules also help maintain healthy and vigorous plants.

Minimizing Traffic over Field

Soil compaction and direct injury to alfalfa crowns and plant tissue occur each time a trip is made over the field. The damage from disease organisms is frequently increased in heavily traveled areas. There has been a recent effort to select alfalfa germplasm with greater tolerance to traffic, but every effort should still be made to reduce compaction.

Movement of Hay, Manure, and Infested Machinery

Disease organisms are often spread from infested to healthy fields by the transport of harvesting equipment, hay, or manure on the farm. Care should be taken to harvest fields that are obviously diseased after harvesting healthy fields, and to thoroughly clean equipment prior to entering healthy fields again.

CONCLUSIONS

Most of the widely known alfalfa diseases were first recognized in hay production fields. As alfalfa becomes more prominent in grazing and cover crop systems, new diseases will likely appear. Significance of pathogens that were once given only passing consideration, such as the members of the “crown rot complex,” may increase. There remains a need to seek out and test alfalfa germplasm for genetic resistance to these pathogens and to investigate the effects of various crop production environments on disease development.

REFERENCES

1. Stuteville, D.L.; Erwin, D.C. *Compendium of Alfalfa Diseases*; APS Press, The American Phytopathological Society: St. Paul, 1990.
2. Hampton, R.O. Diseases caused by viruses and viruses infectious to alfalfa. In *Compendium of Alfalfa Diseases*, 2nd Ed.; Stuteville, D.L., Erwin, D.C., Eds.; APS Press, The American Phytopathological Society: St. Paul, 1990; 51–53.
3. Leath, K.T. Diseases and forage stand persistence in the United States. In *Persistence of Forage Legumes*; Marten, G.C., Matches, A.G., Barnes, R.F., Brougham, R.W., Clements, R.J., Sheath, G.W., Eds.; ASA, CSSA, SSSA: Madison, 1989; 465–478.
4. Fox, C.C.; Berberet, R.; Gray, F.A.; Grau, C.R.; Jessen, D.L.; Peterson, M.A. *Standard Tests to Characterize Alfalfa Cultivars*, 3rd Ed.; North American Alfalfa Improvement Conference: Beltsville, 1991. Available at: <http://www.naaic.org/stdtests/index.html> (accessed April 2005).

Alfalfa Insects: Ecology and Management

John J. Obrycki

James D. Harwood

Department of Entomology, University of Kentucky,
Lexington, Kentucky, U.S.A.

INTRODUCTION

Alfalfa, *Medicago sativa* L., is an important and widely cultivated forage crop throughout the world, representing over 50% of the total U.S. hay production^[1] and over 32 million hectares worldwide. As it withstands a certain degree of pest incidence without significant loss to yield or quality, it is a model crop for the use of biological control and insect pest management.^[1-3] In addition, owing to its relatively high structural diversity compared to that of row crops (e.g., wheat and corn), alfalfa provides a heterogeneous and relatively persistent habitat for natural enemies.

INTEGRATED PEST MANAGEMENT IN ALFALFA

Several comprehensive publications on alfalfa pest management are available in California^[4] and the mid-western U.S.A.^[5] An extensive description of alfalfa integrated pest management (IPM) in North America is available on the web,^[6] but throughout Europe the role of pest management in alfalfa is less frequently documented. Selected researchers have integrated multiple management tactics into management models.^[7,8] The goal of this article is to provide a brief overview of some of the key insect pests of alfalfa and tactics used to suppress them. The primary focus of this entry is on alfalfa IPM in North America.

The literature on the ecology and biology of pests in alfalfa is extensive; in 1991, Steffey and Armbrust^[1] reported that over 9000 literature references were available. One well-documented tactic for alfalfa pest management is the timing of harvesting, which disrupts the life cycles of insect pests, causes direct mortality, and can be used to enhance entomopathogenic fungi.^[7] Selective harvesting or strip harvesting has the potential for manipulating both pest and beneficial species in alfalfa fields.^[9,10] During the late 1990s, several resistant alfalfa cultivars were released, which can have a major role in the suppression of the potato leafhopper in alfalfa fields.^[11]

BIOLOGICALLY BASED SUPPRESSION

Several species of arthropod predators consume insect pests of alfalfa^[12-15] and alfalfa contains a great diversity of predatory arthropods.^[16,17] Two of the most economically important pests of alfalfa are the potato leafhopper *Empoasca fabae* (Harris) and alfalfa weevil, *Hypera postica* (Gyllenhal) in North America.^[1] Several other species of insect herbivores are occasionally abundant in alfalfa, potentially causing economic losses, although the damage to alfalfa tends to be minimal possibly owing to the ability of natural enemies at restricting pest populations.^[18,19] Even if such pests [e.g., *Acyrtosiphon pisum* (Harris)] do not cause significant damage to crops, they are likely to form a significant component of prey for generalist predators and could be important in biological control of key insect pests in alfalfa. Östman and Ives^[20] studied the interactions between the pea aphid and *E. fabae* and reported that predatory nabid bugs aggregated to areas of high aphid density, suggesting that the increase in abundance of aphids would indirectly influence predation on *E. fabae*.

It is the assemblage of predators, rather than the individuals acting alone, that is likely to provide maximum benefit in terms of biological control owing to their diverse feeding habits, temporal variation in abundance, and the fact that they occupy different microhabitat niches. For example, Cardinale et al.^[21] reported that predation levels of the pea aphid in the presence of three species of natural enemies in alfalfa were greater with all three rather than each predator acting alone. An area of research that needs to be pursued in the alfalfa system is postmortem quantitative assessments of predator feeding and trophic interactions in the field (measured by gut-content analysis). The use of these techniques in other agroecosystems has enabled the biological control capacity of natural enemy communities to be quantified^[22-24] and integrated into pest management programs.

Management of the introduced alfalfa weevil in the eastern U.S.A. has developed around the introduction and establishment of several species of parasitic

wasps.^[25] Parasitism caused by these species has resulted in significant declines in population densities of the alfalfa weevil^[26] and reductions in fields requiring insecticide applications.^[25] In addition to these parasitic wasps, an entomopathogenic fungus infects alfalfa weevil larvae and can cause widespread epizootics. Biological control of the alfalfa weevil has been very successful in most areas of the eastern North America; however, the alfalfa weevil is occasionally a problem^[27,28] and remains a persistent pest of alfalfa in areas of the southern U.S.A.

CONCLUSIONS

The precise mechanisms resulting in the depreciation of pest populations could be a result of a number of natural enemy–host interactions. For example, suppression of the alfalfa weevil may include predation of larvae by naturally occurring predators, parasitism by introduced species of parasitic wasps, and infection by entomopathogenic fungi.^[28] Descriptions of alfalfa pest management programs in several regions of North America are included in a comprehensive review^[6] (URL: <http://ipmworld.umn.edu>).

REFERENCES

1. Steffey, K.L.; Armbrust, E.J. Pest management systems for alfalfa. In *CRC Handbook of Pest Management in Alfalfa*; Pimentel, D., Ed.; CRC Press: Boca Raton, FL, 1991; 475–515.
2. Yeargan, K.V. Alfalfa: status and current limits to biological control in the eastern U.S. In *Biological Control in Agricultural IPM Systems*; Hoy, M.A., Herzog, D.C., Eds.; Academic Press: New York, 1985; 521–536.
3. Steffey, K.L.; Armbrust, E.J.; Onstad, D.W. Management of insects in alfalfa. In *Introduction to Insect Pest Management*, 3rd Ed.; Metcalf, R.L., Luckmann, W.H., Eds.; Wiley-Interscience, John Wiley & Sons Ltd.: Hoboken, NJ, 1994; 469–506.
4. Flint, M.L.; Clark, J.K. Integrated pest management for alfalfa hay. University of California Integrated Pest Management Project Publication 3312; 1981; 96 pp.
5. Giles, K.L.; Obrycki, J.J.; Rice, M.E.; Pedigo, L.P.; Barnhart, S.K.; Hogg, D.; DeGooyer, T.A.; Kenney, D.; Munyaneza, J. *Integrated Pest Management of Alfalfa Insects in the Upper Midwest*; ISU Extension and Leopold Center Publication IPM-58, 1999; 48 pp.
6. Flanders, K.L.; Radcliffe, E.B. Alfalfa IPM. In *Radcliffe's IPM World Textbook*; Radcliffe, E.B., Hutchinson, W.D., Eds.; University of Minnesota: St. Paul, MN, 1996. URL; <http://ipmworld.umn.edu>.
7. Brown, G.C.; Nordin, G.L. Evaluations of an early harvest approach for induction of *Erynia* epizootics in alfalfa weevil populations. *J. Kansas Entomol. Soc.* **1986**, *59*, 446–453.
8. Shoemaker, C.A.; Onstad, D.W. Optimization analysis of the integration of biological, cultural, and chemical control of alfalfa weevil (Coleoptera: Curculionidae). *Environ. Entomol.* **1981**, *12*, 286–295.
9. Giles, K.L.; Obrycki, J.J. Reduced insecticide rates and strip-harvesting effects on alfalfa weevil larval populations and prevalence of *Zoophthora phytonomi*. *J. Econom. Entomol.* **1997**, *90*, 933–944.
10. Weiser, L.A.; Obrycki, J.J.; Giles, K.L. Within-field manipulation of potato leafhopper (Homoptera: Cicadellidae) and insect predator populations using an uncut alfalfa strip. *J. Econom. Entomol.* **2003**, *96*, 1184–1192.
11. Sulc, R.M.; van Santen, E.; Johnson, K.D.; Shaeffer, C.C.; Undersander, D.J.; Bledsoe, L.W.; Hogg, D.B.; Willson, H.R. Glandular-haired cultivars reduce potato leafhopper damage in alfalfa. *Agron. J.* **2001**, *93*, 1287–1296.
12. Wheeler, A.G. Studies on the arthropod fauna of alfalfa. 5. Spiders (Araneida). *Can. Entomol.* **1973**, *105*, 425–432.
13. Wheeler, A.G. Studies on the arthropod fauna of alfalfa. 6. Predaceous insects. *Can. Entomol.* **1977**, *109*, 423–427.
14. Barney, R.J.; Armbrust, E.J. List of the insect predators of the alfalfa weevil, *Hypera postica* (Coleoptera: Curculionidae). *Bull. Entomol. Soc. Am.* **1981**, *27*, 241–243.
15. Giles, K.L.; Obrycki, J.J.; DeGooyer, T.A. Incidence and spatial relations of *Hypera postica* predators. *Biol. Control* **1994**, *4*, 170–177.
16. Pimentel, D.; Wheeler, A.G. Species and diversity of arthropods in the alfalfa community. *Environ. Entomol.* **1973**, *2*, 659–668.
17. Yeargan, K.V.; Dondale, C.D. The spider fauna of alfalfa in northern California. *Ann. Entomol. Soc. Am.* **1974**, *67*, 681–682.
18. Losey, J.E.; Ives, A.R.; Harmon, J.; Ballantyne, F.; Brown, C. A polymorphism maintained by opposite patterns of parasitism and predation. *Nature* **1997**, *388*, 269–272.
19. Snyder, W.E.; Ives, A.R. Interactions between specialist and generalist natural enemies: parasitoids, predators, and pea aphid biocontrol. *Ecology* **2003**, *84*, 91–107.
20. Östman, I.; Ives, A.R. Scale-dependent indirect interactions between two prey species through a shared predator. *Oikos* **2003**, *102*, 505–514.
21. Cardinale, B.J.; Harvey, C.T.; Gross, K.; Ives, A.R. Biodiversity and biocontrol: emergent impacts of a multi-enemy assemblage on pest suppression and crop yield in an agroecosystem. *Ecol. Lett.* **2003**, *6*, 857–865.
22. Symondson, W.O.C.; Glen, D.M.; Ives, A.R.; Langdon, C.J.; Wiltshire, C.W. Dynamics of the relationship between a generalist predator and slugs over five years. *Ecology* **2002**, *83*, 137–147.
23. Harwood, J.D.; Sunderland, K.D.; Symondson, W.O.C. Prey selection by linyphiid spiders: molecular tracking

- of the effects of alternative prey on rates of aphid consumption in the field. *Molec. Ecol.* **2004**, *13*, 3549–3560.
24. Harwood, J.D.; Sunderland, K.D.; Symondson, W.O.C. Monoclonal antibodies reveal the potential of the tetragnathids spider *Pachygnatha degeeri* (Araneae: Tetragnathidae) as an aphid predator. *Bull. Entomol. Res.* **2005**, *95*, 161–167.
 25. Day, W.H. Biological control of the alfalfa weevil in the northeastern United States. In *Biological Control in Crop Production*; Papavizas, G.C., Ed.; Allanheld, Osmun: Montclair, NJ, 1981; 361–374.
 26. Kingsley, P.C.; Bryan, M.D.; Day, W.H.; Burger, T.L.; Dysart, R.J.; Schwalbe, C.P. Alfalfa weevil (Coleoptera: Curculionidae) biological control: spreading the benefits. *Environ. Entomol.* **1993**, *22*, 1234–1250.
 27. Harcourt, D.G.; Guppy, J.C. Numerical analysis of an outbreak of the alfalfa weevil (Coleoptera: Curculionidae) in eastern Ontario. *Environ. Entomol.* **1991**, *20*, 217–223.
 28. Giles, K.L.; Obrycki, J.J.; DeGooyer, T.A.; Orr, C.J. Seasonal occurrence and impact of natural enemies of *Hypera postica* (Coleoptera: Curculionidae) larvae in Iowa. *Environ. Entomol.* **1994**, *23*, 167–176.

Animal Breeding

G. Butchaiah

National Biotechnology Center, Rajiv Gandhi College of Veterinary and Animal Sciences,
Pondicherry, India

INTRODUCTION

Many diseases of animals can be controlled by elimination of infected stock, isolation of susceptible individuals, maintenance of hygienic conditions, or vaccination. Drugs are being increasingly used to destroy pathogens or their vectors. Regular use of therapeutics results in resistant forms of pathogens. Further, the use of antibiotics and pesticides to control diseases is prohibitively expensive and could have serious effects on the environment. Moreover, not all diseases can be effectively controlled by these methods. Urgent consideration is therefore needed to develop alternative strategies by raising disease-resistant animals. The identification of animals resistant to specific diseases such as trypanosomiasis, mastitis, colibacillosis, and transmissible spongiform encephalopathies (TSE) promises major benefits on limited but fruitful fronts. The selection of resistant animals for breeding and the application of new molecular biological techniques, particularly the exploitation of genes of the major histocompatibility complex (MHC) and marker-assisted selection (MAS) acting on relevant candidate functional genes or on DNA markers closely linked to quantitative trait loci (QTL), are very much relevant in this process.

GENETIC BASIS OF DISEASE RESISTANCE

Genetic resistance to disease is a common phenomenon in all animal species. Differences in susceptibility of animals to most diseases can be detected. Different pathogens reproduce and propagate themselves by different mechanisms. As such, the genes affecting disease resistance could be equally diverse. No genes conferring universal resistance to diseases have been discovered so far. Indeed, animals resistant to some pathogens could be more susceptible to even closely related organisms, as exemplified by the response of chickens to different *Eimeria* species.^[1] It is shown that the immune system is regulated by sets of genes that control innate (natural) immunity as well as alter the specificity and quality of acquired immunity.

Broadly, genetic resistance can be divided into three main categories: First, genes involved specifically in

host resistance to disease, such as class I and II genes of MHC. Second, genes with structural or metabolic functions that affect disease resistance, e.g., the genes coding for retroviral receptors. Third, genes derived from pathogens themselves may confer resistance either as a consequence of natural processes, as in blocking of E subgroup avian leukosis receptor by endogenous viral envelope genes, or following artificial introduction of genes, as in the equivalent blocking of A subgroup virus in transgenic animals.

In some cases, polymorphism in resistance gene may be selectively maintained, notably among MHC haplotypes. However, data are inadequate to say whether observed polymorphisms are stable or transient as rather little work has been carried out to assess gene frequencies in non-laboratory (commercial) environments. Fortunately, the recently developed techniques for identifying and isolating genes should lead to better understanding of this area because of the possibility of discerning the molecular mode of the resistance genes and to efficiently detect the genes on a commercial scale.

Genetic resistance to a number of infectious diseases caused by bacteria, viruses, and parasites, and production diseases has been demonstrated in various farm animal species (Table 1).

METHODOLOGY OF BREEDING FOR DISEASE RESISTANCE

Exposing unprotected animals to the challenge of the disease in the endemic areas is an expensive and ineffective selection method for disease resistance in farm animals. Whereas, the method of selecting animals for breeding based on natural or artificial tests for resistance to specific diseases shows greater promise for a number of disease conditions. Several gene markers have been identified for use in indirect selection for resistance genes. Although the involvement of two classes of MHC phenotype in disease resistance in cattle is shown, there is evidence only for a few diseases such as mastitis, bovine leukosis, and Theileriosis to show that this knowledge may have practical applications. The results of genome mapping may help in genotype identification. The use of gene transfer to

Table 1 Important diseases/pathogens to which resistance has been demonstrated in animal species

| Species | Disease/pathogen | Mode of resistance (genes associated) |
|----------------|---|---|
| Cattle | Trypanosomiasis | MHC |
| | Theileriosis | MHC |
| | Mastitis | MHC |
| | Tick resistance | – |
| | Bovine leukosis virus | MHC-linked gene |
| | Muscle hypertrophy | DNA marker (associated with susceptibility) |
| | Ketosis | – |
| | Lameness | – |
| Sheep and goat | Haemonchus | MHC/MHC-associated genes |
| | Trichostrongylus | MHC/MHC-associated genes |
| | Footrot | MHC |
| | Ovine cutaneous myiasis | MHC, Non-MHC |
| | Facial eczema | – |
| | Trypanosomiasis | MHC |
| | Maedi/Visna | MHC |
| | Scrapie/TSE | PrP gene |
| Pig | Neonatal diarrhea caused by <i>Escherichia coli</i> | Receptor for <i>E. coli</i> K88 |
| | Malignant hyperthermia | Halothane gene |
| | Coccidiosis | MHC, Non-MHC |
| | Salmonellosis | Non-MHC |
| | <i>Pasteurella multocida</i> | MHC |
| | <i>E. coli</i> | – |
| Chicken | Marek's disease virus | MHC, others |
| | Avian leukosis virus | Virus receptor, MHC |
| | Rous sarcoma virus | Virus receptor, MHC |
| | Newcastle disease virus | – |
| | Infectious bursal disease virus | – |
| | Infectious laryngotracheitis virus | – |
| | Infectious bronchitis virus | – |

produce transgenic animals incorporating specific disease-resistance genes is now a practical feasibility. The relatively high heritability levels (0.3) reported for a range of 15 diseases in Australia and New Zealand show great promise for the future.^[2] However, the establishment of efficient methods for incorporating disease-resistance traits in selection indices is necessary. A high investment in research on relevant genetic correlations and on overall efficacy of resultant breeding schemes is required. However, consideration should be given to the important problem of associated effects of breeding for disease resistance.

Selection Under Challenging Environments

Increased selection pressure for important production traits is often accompanied by increased disease problems. At the same time, selection for enhanced immune responsiveness and disease resistance has often been ignored by animal breeders because of the difficulty in measuring these traits. Actual resistance to individual diseases needs to be measured under an environment including disease challenge. Such testing is prohibitively expensive. The recent

advances in molecular biology and immunology make possible the indirect selection for disease resistance. The MHC genes have a major role in control of disease resistance and all immune functions. Knowledge of the genetic correlations between disease resistance and immune responsiveness and production traits is required for testing and selection for disease resistance and improved immune responsiveness. Antagonistic relationship, if any, between immune response, disease resistance, and production traits might make simultaneous improvement of these traits difficult by conventional breeding and selection methods. Use of MAS or gene transfer methods with MHC gene offer an alternative approach for simultaneous improvement of all traits in such cases.

Application of molecular biological methods

Molecular biology can contribute to enhancing disease resistance of farm animals in two complementary ways—molecular genotyping and gene transfer (transgenesis). Molecular genotyping techniques help in detection of DNA polymorphism, which underlies the

genetic variations between individuals. Such polymorphic marker loci can be used in MAS. For example, selection for a disease-resistance gene for which no direct method of genotyping exists can be performed by selecting appropriate alleles at linked marker loci. Animal genome maps can be derived such that any part of its genome is close to a polymorphic marker locus. The genetic/linkage maps of pigs, sheep, cattle, and poultry being established by various groups worldwide would facilitate the future mapping, manipulation, and cloning of disease-resistance genes.

Genes can now be identified, isolated, and manipulated in the laboratory to derive novel genes for reintroduction into embryos of same or different animals to produce transgenic animals. Although the success rate of producing transgenic animals is low, development of more efficient gene transfer methods for livestock is underway. Such methods of gene transfer include microinjection, calcium phosphate precipitation, retroviral infection, and electroporation. Microinjection of naked DNA into pronuclei of fertilized egg is the most commonly used method for gene transfer. Genes have been transferred into mouse and chicken embryos using retroviral vectors too. The recent demonstrations of site-specific genetic changes by homologous recombination indicates that gene transfer by this route can be used to modify existing genes as well as to add new genes.

The technology of gene transfer is not limited to the use of natural genes alone. Genes can be manipulated in the laboratory to generate entirely new genetic elements. For example, the regulatory sequences from one gene can be fused to the coding sequences of another gene. On introduction of such a hybrid gene into the genome of an animal, the manner and site of expression associated with the regulatory elements will be imposed upon the coding sequences to which they have been fused.

Thus molecular genotyping and gene transfer methods offer complementary means of using biotechnological tools to modify the genetic makeup of the animals to enhance disease resistance.

FUTURE STRATEGY

The present strategy of animal disease control by therapy, vaccination, or reduction of challenge by lower stocking rates may be replaced or aided by breeding for disease resistance in farm animals. This includes selection pressure within conventional breeding schemes on general immunological competence as well as on targeted resistance tests for specific major pathogens. An exciting potential exists in the future for MAS acting on relevant candidate functional genes or on DNA markers closely linked to QTL.

CONCLUSION

Actively selecting and breeding animals adapted to specific pathogens and diseases endemic in their environment, and thus reducing the need for drugs to control these, would seem the most preferred option to control diseases. To yield economically fruitful and environmental benefits as well, highly speculative research and long time period would be required. Production of resistant animals is likely to be expensive and is more of a high-technology procedure. The individual breeder will be tempted to use the safer, short-term pharmaceutical solution to disease control unless persuaded or directed otherwise, particularly as disease-resistant animals are generally not the highest producers.

REFERENCES

1. Bumstead, N.; Millard, B.M.; Barrow, P.; Cook, J.K.A. Genetic basis of disease resistance in chickens. In *Breeding for Disease Resistance in Farm Animals*; Owen, J.B., Axford, R.F.E., Eds.; CAB International: Melksham, U.K., 1991; 10–23.
2. Baker, R.L.; Watson, T.G.; Bisset, S.A.; Vlassoff, A.; Douch, P.G.C. Breeding sheep in New Zealand for resistance to internal parasites: research results and commercial applications. In *Breeding for Disease Resistance in Sheep*; Gray, G.D., Woolaston, R.R., Eds.; Australian Wool Corporation: Melbourne, 1991; 19–32.

BIBLIOGRAPHY

- Albers, G.A.A.; Gray, G.D. Breeding for worm resistance: a perspective. *Int. J. Parasitol.* **1986**, *17*, 559–566.
- Axford, R.F.E., Bishop, S.C., Nicholas, F.W., Owen, J.B., Eds.; *Breeding for Disease Resistance in Farm Animals*; CAB International: Melksham, U.K., 2000.
- Christensen, L.G. Possibilities for genetic improvement of disease resistance, functional traits and animal welfare. *Acta Agric. Scand., A Anim. Sci.* **1998**, *S 29*, 77–89.
- Gibbons, R.A.; Sellwood, R.; Burrows, M.; Hunter, P.A. Inheritance of resistance to neonatal diarrhoea in the pig: examination of the genetic system. *Theor. Appl. Genet.* **1977**, *81*, 65–70.
- Heringstad, B.; Klemetsdal, G.; Ruane, J. *Selection for Mastitis Resistance in Dairy Cattle—A Review with Special Consideration of the Situation in the Nordic Countries*, 47th Annual Meeting of the EAAP, Lillehammer, Norway, August 25–29, 1996; 1996.
- International Livestock centre of Africa (ILCA). *Trypanotolerant Livestock in West and Central Africa*; ILCA: Addis Ababa, Ethiopia, 1979; Vol. 1, Monograph No. 2.
- Lewin, R.A. Disease resistance and immune response genes in cattle: strategies for their detection and evidence of their existence. *J. Dairy Sci.* **1989**, *72*, 1334–1348.
- Raadsma, H.W. Genetics of resistance to fleece rot and footrot. In *Proceedings Australian Association of Animal Breeding and Genetics*; 1991; Vol. 9, 67–72.

Aphid and Aphid-Borne Virus Management: Squash Disease Control

Charles G. Summers

Department of Entomology, University of California, Parlier, California, U.S.A.

James J. Stapleton

Statewide IPM Project, UC Kearney Agricultural Center, Parlier, California, U.S.A.

Jeffrey P. Mitchell

Kearney Agricultural Center, University of California, Parlier, California, U.S.A.

INTRODUCTION

Summer squash (*Cucurbita pepo* L.) is susceptible to several aphid-borne viruses, including cucumber mosaic virus (CMV), zucchini yellow mosaic virus (ZYMV), and watermelon mosaic virus (WMV). All are transmitted in a nonpersistent manner by several species of aphids. Plants may become infected with one or all of the viruses and those infected by multiple viruses suffer more damage than those infected with only one virus.

OVERVIEW

Infected plants become discolored and the leaves misshapen (Fig. 1). The fruit is distorted and mottled and is not marketable (Fig. 2). Insecticides offer little relief, as the viruses are acquired and transmitted by the vector within 20 to 30 sec, usually before the aphid vector obtains a lethal insecticide dose.^[1] Insecticides may actually enhance virus spread.^[2] Ultraviolet (UV) reflective plastic mulch has been used successfully to delay and reduce the incidence of aphid-borne virus diseases in many crops.^[3-5] Wheat straw has also been found to reduce the incidence of aphid infestation.^[6] Burton and Krenzer^[7] determined that straw mulch decreased the incidence of alate aphid landings. Bwy Jones, and Proudlove^[8] found a decrease in the incidence of CMV in narrow-leaved lupine grown over straw, and Jones^[9] observed reduction in the incidence of bean yellow mosaic virus in lupines planted in cereal straw.

REFLECTIVE PLASTIC AND WHEAT STRAW MULCH REDUCES APHID ALIGHTING AND DELAYS VIRUS INFECTION

Studies were conducted to determine whether reflective plastic and wheat straw mulch could be successfully

used to manage aphid-borne viruses. These strategies were compared to a conventional insecticide treatment, imidacloprid injected into the soil as a preplant application, and a bare soil control of no mulch and no insecticide.

Both the reflective plastic and the straw mulch worked well in repelling alate aphids (Fig. 3). The number of aphids per leaf was lower in these plots than in either the imidacloprid or control plots. The UV reflective plastic mulch and the straw mulch provided equal protection against aphid-transmitted viruses (Fig. 4). Six weeks after planting (the fourth sample), the incidence of virus-infected plants growing over these two mulches was <30%. On the same sample date, the incidence of virus-infected plants in the imidacloprid-treated plots and the bare soil control ranged from 60% to 75%. These two treatments did not differ from each other. The UV reflective plastic and the straw mulch delayed the appearance of virus-infected plants for 3 to 4 weeks beyond that observed in the bare soil and imidacloprid-treated plots. Similar results were obtained in a second study, although the virus disease incidence was lower. By the time the sixth sample was taken, the incidence of virus-infected plants in the two mulched plots was <10%, whereas in the imidacloprid and control plots, it approached 30% to 40%. Imidacloprid failed to provide any practical protection; the incidence of virus infection in these plots did not differ significantly from that in the bare soil control plots. Plants growing over UV reflective plastic and straw mulch produced higher yields than those growing over the imidacloprid and the untreated control plots. In 2000 (11 harvests), the equivalent of 46,647, 47,471, 22,529, and 4118 lb/acre were harvested from the reflective plastic, wheat straw mulch, imidacloprid, and control treatments, respectively. In 2001, extrapolated yields (11 harvests) averaged 14,647, 24,235, 9059, and 2353 lb/acre from the reflective plastic, wheat straw mulch, imidacloprid, and control treatments, respectively.

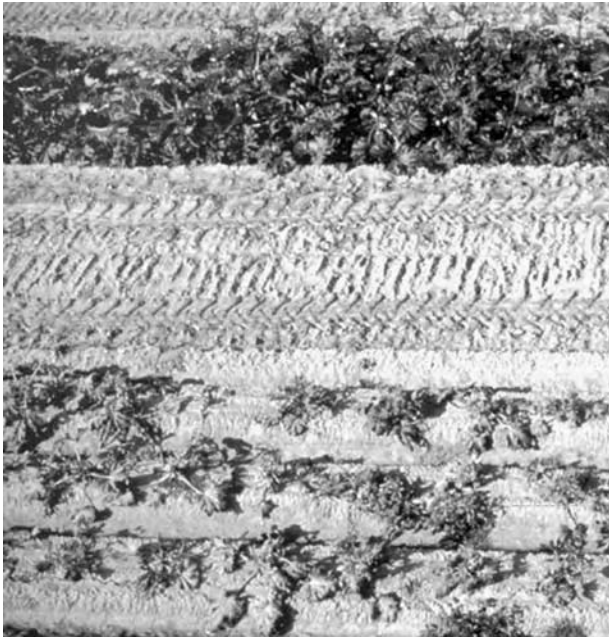


Fig. 1 Damage caused by multiple virus infections of zucchini squash plants growing over bare soil (bottom) compared to uninfected plants growing over reflective plastic mulch (top).

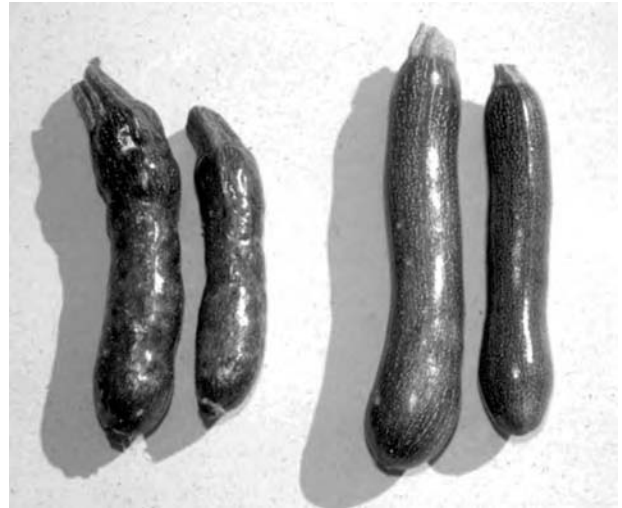


Fig. 2 Zucchini squash fruit infected with one or more viruses (left) compared to noninfected fruit (right).

Grower Trials

Reflective plastic mulch was compared to conventional production (squash planted on bare soil) in two

10-acre commercial squash fields, identified by the grower names, Kubo and Solis. Reflective plastic was laid over the planting beds in one-half of each field, and in the remaining half, seeds were planted over bare soil. Fields were evaluated weekly for virus incidence. Yields (11 harvests) were taken from the Kubo field.

The incidence of virus disease was higher in the Kubo field than in the Solis field. The reflective plastic mulch reduced the number of plants presenting

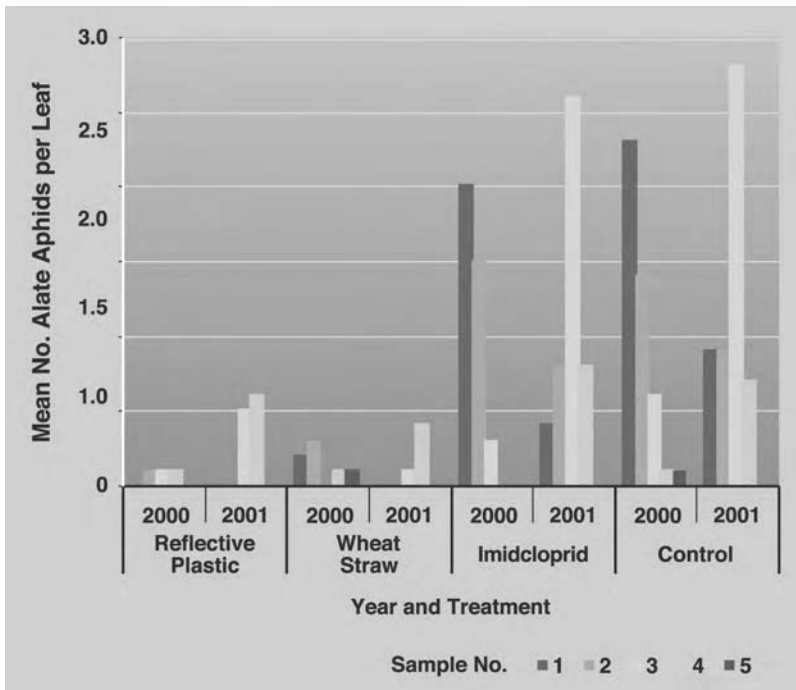


Fig. 3 Alate aphid population on zucchini squash plants.

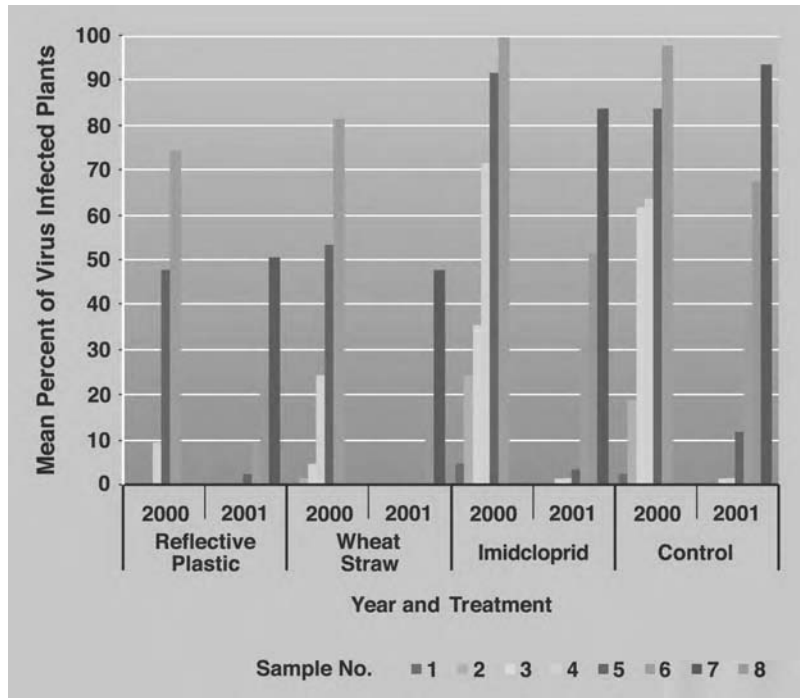


Fig. 4 Incidence of virus-infected plants on zucchini squash.

symptoms in both. In the Kubo field, no virus-infected plants growing over the reflective plastic were observed until 2 October, and none were detected in the Solis field until 10 October (Fig. 5). In the Kubo field, disease incidence reached 100% in the unmulched half by 10 October, while the disease incidence in the mulched half remained under 20% for the entire

season. In the Solis field, the disease incidence approached 60% in the unmulched half but remained below 10% in the mulched half throughout the season. The yield of marketable fruit from the mulched portion of the Kubo field averaged 53,166 lb/acre, while the yield from the unmulched portion of the field averaged 25,177 lb/acre.

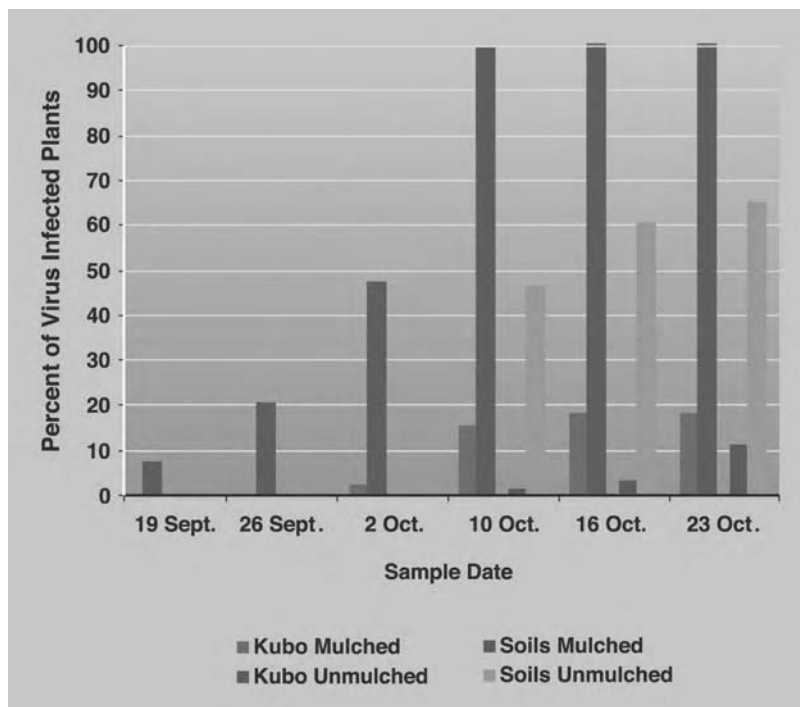


Fig. 5 Incidence of virus infected plants in two commercial zucchini squash fields growing over reflective plastic mulch and bare soil.

MECHANISM OF UV REFLECTIVE PLASTIC MULCH AND WHEAT STRAW MULCH IN REPELLING APHIDS

Ultraviolet reflective mulches reflect shortwave UV light,^[10,11] which confuses and repels incoming alate aphids, thus reducing their incidence of alighting on plants.^[12] To determine the amount of UV radiation being reflected from the various mulches, spectral energy distributions from the mulches were performed at 2-nm intervals between the wavelengths of 300 and 700 nm.

The reflective plastic mulch was superior to both straw and bare soil in reflecting UV wavelengths. The plastic reflected 86% of the incoming UV compared to the ambient. The reflected UV serves to repel alate aphids.^[12] Across the UV spectrum there was no difference between the straw mulch and the bare soil. The reflective plastic mulch reflected 94% of the incoming photosynthetically active radiation (PAR), 400 to 700 nm, compared to the ambient, whereas the straw mulch reflected 85%. Bare soil reflected only 41% of incoming PAR. Plant growth and fruit production were definitely linked to reduced levels of insect and disease pressure, but it is also likely that PAR reflected back into the canopy helped contribute to increased growth, development, and production.

CONCLUSION

Non-persistent aphid-borne viruses of squash can be successfully managed using plastic reflective or wheat straw mulch. Both techniques resulted in a reduced incidence of virus disease. Yields of marketable fruit increased approximately 10-fold in small plot trials and twofold in a commercial squash field. Acceptable yields can be obtained without the use of chemical pesticides. The plastic mulch works by reflecting a high percentage of UV light back into the sky, thus repelling incoming alate aphids. The mechanism involved in the repulsion of aphids by the wheat straw mulch remains to be determined.

REFERENCES

1. Gibson, R.M.; Rice, A.D. Modifying aphid behaviour. In *Aphids: Their Biology, Natural Enemies and Control*; Minks, A.K., Harrewijn, P., Eds.; Elsevier: Amsterdam, 1989; 209–224.
2. Ferro, D.N.; Mackenzie, J.D.; Margolies, D.C. Effect of mineral oil and a systemic insecticide on field spread of aphid-borne maize dwarf mosaic virus in sweet corn. *J. Econ. Entomol.* **1980**, *73*, 730–734.
3. Stapleton, J.J.; Summers, C.G. Reflective mulches for management of aphids and aphid-borne virus diseases in late-season cantaloupe (*Cucumis melo* L. var. *cantalupensis*). *Crop. Prot.* **2002**, *21*, 891–898.
4. Summers, C.G.; Stapleton, J.J. Management of vegetable insects using plastic mulch: 1997 season review. *U.C. Plant Protect. Q.* **1998**, *8* (1&2), 9–11.
5. Summers, C.G.; Stapleton, J.J. Management of aphids, silverleaf whiteflies, and corn stunt leafhoppers using reflective plastic mulch and insecticides: 1998 season review. *U.C. Plant Protect. Q.* **1999**, *9* (1), 2–7.
6. Liewehr, D.J.; Cranshaw, W.E. Alate aphid traps capture over different background colors and different background patterns. *Southwestern Entomol.* **1991**, *16*, 13–18.
7. Burton, R.L.; Krenzer, E.G., Jr. Reduction of greenbug (*Schizaphis graminum*) (Homoptera: Aphididae) populations by surface residues in wheat tillage studies. *J. Econ. Entomol.* **1985**, *78*, 390–394.
8. Bwyne, A.M.; Jones, R.A.C.; Proudlove, W. Effects of different cultural practices on spread of cucumber mosaic virus in narrow-leafed lupins (*Lupinus angustifolius*). *Aust. J. Agric. Res.* **1999**, *50*, 985–996.
9. Jones, R.A.C. Effect of mulching with cereal straw and row spacing on spread of bean yellow mosaic potyvirus into narrow-leafed lupins (*Lupinus angustifolius*). *Ann. Appl. Biol.* **1994**, *124*, 45–58.
10. Harpaz, I. Nonpesticidal control of vector-borne viruses. In *Pathogens, Vectors and Plant Diseases: Approaches to Control*; Harris, K.F., Maramorosch, K., Eds.; Academic Press: New York, 1982; 1–21.
11. Loebenstein, G.; Raccach, B. Control of nonpersistently transmitted aphid-borne viruses. *Phytoparasitica* **1980**, *8*, 221–235.
12. Kring, J.B. Flight behavior of aphids. *Annu. Rev. Entomol.* **1972**, *17*, 461–492.

Asian Longhorned Beetle: Ecology and Control

Ann E. Hajek

Department of Entomology, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

The Asian longhorned beetle (ALB), *Anoplophora glabripennis*, is a striking large black beetle with long antennae with white stripes and irregularly scattered white spots on its wing covers (elytra). Bodies of adult beetles are usually 17–39 mm long, but females are normally slightly larger than males. Antennae of males are often nearly two times the body length while the antennae of females are usually less than 1.5 times the body length.^[1] In China, where these beetles are native, the large members of this beetle family are often referred to as “sky water buffaloes” because of their long antennae. However, in the case of ALB, behind this poetic name is a serious pest that can attack and kill apparently healthy hardwood trees (Fig. 1).

DISTRIBUTION AND HISTORY

ALB is native to mainland China and Korea.^[2] In China alone, this beetle is distributed over an area approximately the size of North America, ranging from the northeastern provinces, formerly referred to as Manchuria, to the drier province of Gansu to the east, and south to the province of Sichuan. It is hypothesized that the original range of this beetle included eastern China, extending north into Korea, but *A. glabripennis* was never considered a pest in the natural forests in China or Korea. Studies of naturally occurring low-density populations of ALB in natural forests of Korea have shown that this day-flying beetle predominantly attacks maple trees (*Acer* spp.) along streams and, therefore, is specialized at exploiting the edges of forested habitats.

The distribution and abundance of this beetle increased dramatically in China due to human manipulations. In the 1960s and 1970s, the Chinese government supported a widespread reforestation program to plant millions of trees, principally in the north, to replace those that had been cut to increase the area available for farming.^[2] Millions of trees were planted as hedgerows, along roadsides, as shelter belts along farmers' fields, as street and park trees in cities, and in plantations for pulp production. Tree species chosen for planting were all poplars, which grow quickly, but the diversity of species planted was minimal. *Populus*

dakhuensis was the most commonly planted species. Poplars are within the host range of ALB. Moreover, the poplars planted during this “greening” campaign were probably especially attractive and vulnerable to ALB because the species planted were not well-adapted to many of the planting sites, especially those areas receiving little rainfall. Many trees were water-stressed and longhorned beetles are generally thought to preferentially attack water-stressed trees. In addition, with a virtual monoculture of acceptable trees covering a huge area, populations of this beetle have increased phenomenally since the late 1970s. Thus, ALB became a serious tree pest in northern China. Populations of these beetles increased in plantings of susceptible trees, leaving the trees dead and earning their nickname “the forest fire without smoke.” Chinese scientists have estimated that *A. glabripennis* has caused the loss of hundreds of millions of trees, including some susceptible trees that were not infested but were cut down to prevent increase and spread of the pest.

With such large populations of ALB in China, it was only a matter of time before this beetle was accidentally introduced elsewhere. In 1996, ALB was first found in North America, in Brooklyn adjacent to Manhattan, NYC, having arrived in untreated, infested solid wood packing material used in international cargo. Two years passed before ALB was also found in Chicago, followed by detection in New Jersey in 2002 and in Toronto in 2003. This beetle has also now been detected in Austria (2001), France (2003), and Germany (2004). As evidenced in China, this beetle has enormous destructive potential. Using a worst case scenario for the United States, in 2001 potential losses were estimated at over \$600 billion.^[3] At present, all known infestations in North America occur in urban/suburban trees and not natural forests. Thus, the areas in North America infested by ALB have been constructed or manipulated by humans. These tree plantings are often water-stressed and have abundant edge habitat. ALB appears to be particularly well adapted to exploiting the current infested area.^[4]

BIOLOGY

Asian longhorned beetles usually have a one year life cycle, but sometimes two years are required for



Fig. 1 Asian longhorned beetle adult. Photo taken in Inner Mongolia, China by A.E. Hajek.

development. Larvae of ALB develop within the wood of living trees. During summer, newly formed adults chew an emergence hole and emerge from trees. Emergence occurs over a prolonged period and is affected by climate. Emergence has been reported as beginning from April to June, and continuing throughout the summer and fall, with the last adults recorded in the field up to December. Both sexes require a period of one to two weeks before they are sexually mature and, during this time, they feed on the tender bark of twigs and branches, the midribs of leaves and petioles. Upon maturity, beetles of both sexes are attracted to trees by tree-produced volatiles and, once a male and female are on the same tree, the male responds visually to the other ALB and then recognizes females using a contact pheromone.^[5] After mating, females chew into the cambial layer under the bark and then turn around to lay an egg under the inner bark. Adult females can live from one to over six months, and total egg-laying has been shown to range from 30 to 178 viable eggs from individual females fed on different tree species at different temperatures.^[6,7] Eggs hatch within a few weeks after being laid and larvae feed by tunneling under the bark as they grow, and then move into the interior of the wood (the xylem) once they are larger. Larvae are very solitary and tunnels of

individuals do not merge, although many individuals can be developing in the same area within the tree. The shavings produced by large larvae while tunneling in the center of the tree are pushed out of the trees and serve as one of the first, more readily observed indications that ALB larvae are within the tree. Larvae tunnel upward approximately 10–30 cm and become 50–60 mm long before pupation. Pupation occurs in a chamber near the bark surface and adults emerge through 6–18 mm diameter circular holes.

Although ALB larvae seem to be protected by developing within trees and adults have a thick protective integument, survival of beetles is not very high. A study in China estimated 35–38% survival from egg to adult.^[8] The major causes of mortality were predators (including woodpeckers), parasitoids and fungal and bacterial pathogens, unexplained failure of smaller larvae to continue developing, and low temperatures. In North America, the natural enemies that evolved with ALB are not present, yet survival values that have been calculated still range from 7–30%. Although these survival levels seem low, they can still result in increasing ALB populations.

Although the dynamics of infestation are presently being worked out, it seems that when ALBs first attack a tree, they oviposit where there is thinner bark, often in branches in the upper canopy. Eggs can be laid in branches as small as 3 cm in diameter. Numerous eggs can be laid in the same branch by a female. Once the first generation of beetles emerges as adults, the tendency is for most adults to stay and reattack the same tree. On trees that are reattacked, first the upper branches and then the tree crown dies and, in response, successive generations of beetles lay their eggs in branches further down, closer to the trunk and eventually in the tree trunk itself, until the tree dies. These large-bodied beetles are reluctant flyers and when placed on host trees, many females remained on the same tree or did not go far.^[9] When beetles have been forced to fly, many were recaptured within 200 m of the release site, although a few were captured as far as 2600 m away,^[10] suggesting the potential of a small percentage of these beetles to fly longer distances, although most dispersers probably remain in the area near their natal tree.

Asian longhorned beetles do not infest only poplars but have much broader host preferences. In China, this species primarily infests poplars (*Populus*), willows (*Salix*), elms (*Ulmus*) and maples (*Acer*), but larvae are known to develop in a total of 24 species of deciduous trees. In North America, ALB has been reported from at least 18 species of deciduous trees in 12 genera.^[2] Many of the tree species that have been attacked are commonly used as urban street trees, e.g., elms (*Ulmus*), ashes (*Fraxinus*), willows (*Salix*), poplars (*Populus*), birch (*Betula*) and horsechestnut (*Aesculus*), but the highly preferred species of greatest concern are

the maples (*Acer*), which are very widely planted along city streets and in parks. Concern about the occurrence of ALB in northeastern North America was fueled by the fact that these beetles attack maples, a very common genus of trees in northeastern forests that is also important as the source of traditional maple syrup.

DETECTION AND CONTROL

ALB is an invasive species with great potential to wreak havoc in North American forests as well as urban and suburban plantings. Ecological niche modeling has shown that ALB could invade most of eastern North America but only limited areas of western North America.^[11] However, thankfully, infestations to date in the United States and Canada have been found only in urban and suburban areas and not in native forests. In response, the goal of both United States and Canadian governments has been to eradicate this beetle. However, this is not an easy beetle to control. Adults are present over a long time period, often high in tree canopies, and larvae are difficult to detect when they are within trees. The major control practice utilized in China has been to cut down all susceptible poplars and replant with resistant hybrids. However, this option seems less feasible in the urban and suburban landscape of North America.

In North America, it was quickly learned that ALB is very difficult to detect; in fact, ALB was probably present in the United States for up to 10 years before it was first detected in 1996. At present, in infested areas, trees are surveyed for beetles within a 0.5 mile radius from each infestation point. Present detection practices are composed of visual inspection of individual trees in infested areas, specifically looking for emergence holes, oviposition scars, and wood shavings from tunneling by larger larvae, sap flow from oviposition sites and flagging of upper branches in otherwise healthy trees. However, survey methods are time and labor-intensive and are often not as efficient as desired. When found, infested trees are cut and chipped and subsequently replaced with non-host trees. Quarantine boundaries are adjusted based on locations of infested trees so that infested wood is not moved out of the regulated areas. In the United States, by the end of 2004, to control ALB, 6187 trees had been removed from New York, 1553 from Illinois and 548 from New Jersey.

Because adult ALB in North America are neither abundant nor easily detectable, and infestations are in urban areas, spraying tree canopies with synthetic chemical insecticides is not an option and alternative types of control are required. In most infested areas in the United States, tree trunks have been injected with the beetle-active chemical insecticide imidacloprid or soil is injected and this chemical is taken up systemically

by trees. This treatment can kill smaller larvae feeding directly under the bark, larger larvae in the centers of branches and trunks, as well as adults feeding on the outer bark of twigs if they receive a high enough dose. However, chemical analyses have demonstrated that imidacloprid is often not evenly dispersed throughout trees when injected into trunks.^[12] Perhaps more importantly, imidacloprid also acts as an anti-feedant,^[13] so adults feeding on a treated tree would be repelled and larvae may stop feeding and starve to death if not killed. Using imidacloprid to prevent ALB attacks makes most sense if all the valuable trees in an area are treated, as in an eradication program. Between 2000 and 2004, over 500,000 trees were treated with imidacloprid in the infested areas of New York, Illinois and New Jersey. In contrast, the localized, smaller infestation in Toronto has been treated by cutting down all susceptible trees in the quarantine area.

Because trunk injections of imidacloprid may not be 100% effective and there is the potential for beetles to develop within non-injected trees, additional control strategies are being developed. Fiber bands containing cultures of an insect-specific fungal pathogen are used for control of a closely related beetle that is an orchard pest in Japan and this methodology is being adapted for control of ALB. Bands attached around tree trunks and branches target wandering adults that become inoculated with fungal spores when contacting the bands. The spores germinate, the fungus penetrates through the beetle cuticle, and then proliferates, killing the adult beetle. Adult females walking on bands either do not reproduce before death or lay fewer eggs than healthy beetles before dying. Adults that have walked across a band can also inoculate others during mating. Field studies in China have shown that in plots with fungal bands on trees, adults died sooner than in untreated plots and egg-laying declined drastically.^[14] The USDA has purchased these bands for application in areas where adult beetles are known to live.

ACKNOWLEDGMENTS

We thank E. R. Hoebeke, M. Keena, T. Poland and R. Shanley for sharing unpublished data and for providing comments on this summary and Alphawood Foundation for support.

REFERENCES

1. Hajek, A.E.; Curtiss, R.T.; Lieberr, J.K. Characters differentiating male from female *Anoplophora glabripennis* (Motschulsky) (Coleoptera: Cerambycidae). *Proc. Entomol. Soc. Wash.* **2005**, *106* (4), 928–931.

2. Lingafelter, S.W.; Hoebeke, E.R. *Revision of Anoplophora (Coleoptera: Cerambycidae)*; Entomological Society of Washington: Washington, DC, 2002.
3. Nowak, D.J.; Pasek, J.E.; Sequeira, R.A.; Crane, D.E.; Mastro, V.C. Potential effect of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) on urban trees in the United States. *J. Econ. Entomol.* **2001**, *94*, 116–122.
4. Williams, D.W.; Lee, H.P.; Kim, I.K. Distribution and abundance of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) in natural *Acer* stands in South Korea. *Environ. Entomol.* **2004**, *33* (3), 540–545.
5. Zhang, A.; Oliver, J.E.; Chauhan, K.; Zhao, B.; Xia, L.; Xu, Z. Evidence for contact sex recognition pheromone of the Asian Longhorned Beetle, *Anoplophora glabripennis* (Coleoptera: Cerambycidae). *Naturwissenschaften* **2003**, *90*, 410–413.
6. Keena, M.A. *Anoplophora glabripennis* (Coleoptera: Cerambycidae) fecundity and longevity under laboratory conditions: comparison of populations from New York and Illinois on *Acer saccharum*. *Environ. Entomol.* **2002**, *31* (3), 490–498.
7. Smith, M.T.; Bancroft, J.; Tropp, J. Age-specific fecundity of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) on three tree species infested in the United States. *Environ. Entomol.* **2002**, *31* (1), 76–83.
8. Zhao, R.; Lu, Z.; Lu, X.; Wu, X. Life table study of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) natural populations. *J. Beijing For. Univ.* **1993**, *15* (4), 125–129.
9. Williams, D.W.; Li, G.; Gao, R. Tracking movements of individual *Anoplophora glabripennis* (Coleoptera: Cerambycidae) adults: application of harmonic radar. *Environ. Entomol.* **2004**, *33* (3), 644–649.
10. Smith, M.T.; Tobin, P.C.; Bancroft, J.; Li, G.; Gao, R. Dispersal and spatiotemporal dynamics of Asian Longhorned Beetle (Coleoptera: Cerambycidae) in China. *Environ. Entomol.* **2004**, *33* (2), 435–442.
11. Peterson, A.T.; Scachetti-Pereira, R.; Hargrove, W.W. Potential geographic distribution of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) in North America. *Am. Midl. Nat.* **2004**, *151* (1), 170–172.
12. Poland, T.; Haack, R.A.; Petrice, T.R.; Miller, D.L.; Bauer, L.S.; Gao, R. Field evaluations of systemic insecticides for control of the Asian Longhorned Beetle, *Anoplophora glabripennis* (Coleoptera: Cerambycidae), in China. *J. Econ. Entomol.* **2006**, *99* (2), 383–392.
13. Poland, T.; Haack, R.A.; Petrice, T.R.; Miller, D.L.; Bauer, L.S. Laboratory evaluation of the toxicity of systemic insecticides for control of the Asian Longhorned Beetle, *Anoplophora glabripennis*, and the cottonwood borer, *Plectrodera scalator* (Coleoptera: Cerambycidae). *J. Econ. Entomol.* **2006**, *99* (1), 85–93.
14. Hajek, A.E.; Huang, B.; Dubois, T.; Smith, M.T.; Li, Z. Field studies of control of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) using fiber bands of the entomopathogenic fungi *Metarhizium anisopliae* and *Beauveria brongniartii*. *Biocontrol Sci. Technol.* **2006**, *16* (4), 329–343.

Asian Longhorned Beetle: Invasion on North American Urban Forests

E. Richard Hoebeke

Department of Entomology, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

Since 1996, the U.S. Department of Agriculture (USDA) has been waging a battle to eradicate an exotic beetle that could threaten one of the country's most valuable natural resources—its trees. This tree-infesting beetle is called the Asian long-horned beetle, *Anoplophora glabripennis* (Motschulsky) (Fig. 1). Forest entomologists believe that this alien invader could potentially become the worst ecological disaster that North American forests have ever witnessed. In fact, the USDA and its regulatory arm, the Animal and Plant Health Inspection Service (APHIS), whose mission it is to protect American agriculture and forestry from foreign pests, have declared that this beetle, if it were to become established here, “could turn into the gypsy moth of the 21st century, destroying millions of acres of America's treasured hardwoods, including national forests and backyard trees.”^[1] Lethal to a wide range of hardwood trees, and especially maples, Asian long-horned beetles threaten everything from backyard, park, and city street trees, to the maple sugar industry, to tourism in fall-foliage regions of the northeast. These invaders are native to China and Korea. In China, they are widespread tree pests, particularly in poplar tree plantations. Thus far, the beetles have infested standing trees in only a few parts of the country—the New York City–Long Island area and several parts of Chicago.

CITIES UNDER SIEGE

“Wanted: The Asian Long-Horned Beetle” posters (Fig. 2) are commonly encountered these days—affixed to billboards, telephone poles, and street lamps, and in the news media—throughout the New York City and Chicago metropolitan areas, whose urban landscapes have been under attack by this pest. More than 7000 trees in and near these metropolitan centers have been removed and destroyed so far by federal and state regulators in an attempt to prevent its unimpeded spread. In addition to these standing tree infestations, the beetles have also been intercepted in more than 26 other

cities in at least 14 states. The majority of intercepted beetles have been found by federal inspectors at ports of entry and in high-risk importer warehouses. A close inspection of solid wood packing materials—such as crating, pallets, dunnage, wood spools, wood blocks, and large dimensional wood skids, all used in the international trade industry to stabilize packaged products in transit—has uncovered stealth introductions of Asian long-horned beetles and other exotic wood-boring beetles. The Asian long-horned beetle now joins at least 400 other species of exotic insects in North America, which have become naturalized on native and introduced woody plants of our forests, parks, and urban landscapes.^[2]

SOLID WOOD PACKING MATERIALS: BEWARE

No one today doubts that the Asian long-horned beetle most likely gained entry into North American landscapes through the extensive use of solid wood packing materials. Urban landscapes, composed of millions of street trees, and native hardwood forests are at risk and are highly vulnerable to pests that hitchhike on wood packaging. In fact, data compiled by the USDA-APHIS show that 90% or more of the insects that are potential forest pests reach the country on crates, pallets, or other forms of solid wood packing material, which is often untreated green lumber and of low quality because it is insect-infested. Between 1995 and 1998, 97% of the potential forest pests detected by USDA port inspectors were associated with solid wood packing material.^[3] According to the U.S. General Accounting Office,^[4] U.S. imports rose by more than 50% between 1990 and 1997, and each year over 400,000 planes and 53,000 ships (Fig. 3A) enter our country,^[5] carrying 24–30 million shipping freight containers (Fig. 3B) filled with goods and merchandise from abroad. According to some USDA estimates, as much as half of the ocean-borne shipments and around 10% of the airborne shipments are packaged in wood.^[3] This wood packaging, a mere by-product of trade, is dismantled and often discarded outside warehouses into large refuse piles (Fig. 3C). Any wood-boring



Fig. 1 A female Asian long-horned beetle (foreground) and larval feeding damage to a trunk of Norway maple (background). (Photo by C. Harrington.)

insect that escapes this infested wood seeks out suitable host trees in a newfound habitat.

GROUND ZERO—WHERE IT ALL BEGAN

It is speculated that the Asian long-horned beetle arrived in Brooklyn as a stowaway, perhaps buried deep in blocks of wood used to stabilize a shipload of sewer pipes from China. Although the beetles were first discovered in the summer of 1996, it is thought that they were probably in Brooklyn as long as a decade or more before anyone ever noticed them. But, by August 1996, the pests had already saturated the northern Brooklyn community of Greenpoint^[6] to the extent that many of the street trees there were dying. A resident of Greenpoint noticed that several Norway maples lining McGuinness Avenue were heavily damaged and many were dying. Trunks of these street trees, 6–8 in. in diameter, had numerous gaping, circular holes (about 3/8–3/4 in. in diameter). At the base of the trunks along the sidewalks were large accumulations of coarse sawdust. Copious sap flows were coming from crescent-shaped notches in the bark, and there was substantial branch dieback. As it turned out, these were all physical signs of attack by the Asian long-horned beetle. Once the beetle had been identified to species and determined to be an exotic, invasive invader, a deluge of press coverage followed, which was attributed to its discovery about a month later in the Amityville area of Long Island. Two years later, in July 1998, Asian long-horned beetle was found, quite by accident, infesting trees in Ravenswood, Chicago.

Again, as a result of an outpouring of media attention, local citizens were responsible for the discovery of two additional, but smaller infestations in outlying areas of Chicago.

BATTLE ZONES ESTABLISHED AND FORTIFIED

Federal, state, and city cooperators in New York and Illinois, as well as the U.S. Forest Service, have promoted eradication activities by imposing quarantines, by conducting surveys around confirmed sites, and by removing and destroying infested trees. Even with these efforts, the beetles continue to spread in these metropolitan areas. Current priorities include the completion of surveys to delimit the extent of the infestations in both states by using the most effective methodology, including tree climbers and the use of bucket trucks.^[7] Information from these intensive surveys has been and continues to be used to determine boundaries of regulated areas. At present, several distinct outbreak areas can be identified in both Illinois and New York, each of which is quarantined. In Illinois, there is one large, generally infested area in Ravenswood, with several smaller satellite outbreaks in Addison, Summit, Kilburn Park, Park Ridge, and near O'Hare International Airport. New York has two generally infested areas in Brooklyn–Queens and in central Long Island (Amityville area); satellite outbreaks are found in Bayside, Flushing, Flushing Meadows Corona Park, Upper and Lower East Side Manhattan, and the Murray Hill area. The only known



Fig. 2 A “Wanted” poster for the Asian long-horned beetle as it appeared in a 1998 daily edition of the Chicago Tribune newspaper. (From the USDA-APHIS, Marketing and Regulatory Programs, Legislative and Public Affairs.)

satellite outbreak on Long Island is near Islip. To date, over 5500 infested trees have been removed in New York, while in the Chicago area over 1500 infested trees have been destroyed.^[7]

IMPACT ABROAD

Before 1970, there was little mention of *A. glabripennis* in the literature. But in the past 30 years, the Asian long-horned beetle has become one of the most important pests of poplar trees in mainland China. By the mid to late 1970s, the Chinese Government launched a reforestation program to replace many of the native trees lost when vast acreages were cleared to create farmlands for a growing population. Government

officials promoted the broad-scale planting of poplars because of their ability to grow quickly in the harsh climates of northern China. Before long, the landscape became a monoculture comprising one or two native species of *Populus*, most of which were highly susceptible to attack by *A. glabripennis*. In addition to poplars being planted around agricultural fields as shelter belts, in cities along streets and in parks, it also has been the wood of choice for the manufacture of crating and pallets used in the international trade industry. As much as 45% of the poplar plantations in China have been severely damaged by various long-horned beetles, and particularly *A. glabripennis*.

In rural areas of northern China, the Asian long-horned beetle is known among the locals as “the forest fire without smoke” because of its devastating effects on the forests. While the Asian long-horned beetle has proliferated virtually uncontrolled in northern China, estimates of its impact there vary widely. Chinese scientists indicate that hundreds of millions, if not billions, of trees have been either infested by the Asian long-horned beetle, or else cut down to prevent its spread. There are claims that the Asian long-horned beetle has caused in excess of US\$100 million in damage and has infested half of the trees within a 5000-square mile area. Media accounts in China have told how 142 million trees were destroyed by the Asian long-horned beetle in one province alone over a 6-year period, and that over 50 million trees were cut down from 1991 to 1993 in Ningxia Province, resulting in losses estimated at US\$37 million.^[8] Some studies claim that the Asian long-horned beetle has dispersed into more than 240 cities in the five hardest hit provinces of northern China, affecting nearly 600,000 acres.

POTENTIAL ECOLOGICAL DISASTER AT HOME

The true impact of the Asian long-horned beetle in the United States has yet to be determined. In spite of the fact that this exotic forest pest may be confined to New York City and Chicago and that the quarantine zones have indeed limited its spread to the outside, overall efforts to date to eradicate this wood-boring beetle have cost federal, state, and local regulatory agencies millions of dollars. According to a recent Forest Service/APHIS study, the total value of tree resources at risk in the cities of New York and Chicago alone is approximately US\$2.3 billion and US\$1.2 billion, respectively. The estimated potential national impact of the Asian long-horned beetle, if every urban center in the United States becomes infested, is a loss of about 35% of the canopy cover, 30% of the trees (approximately 1.2 billion trees), and US\$669 billion dollars in compensatory value.^[9]

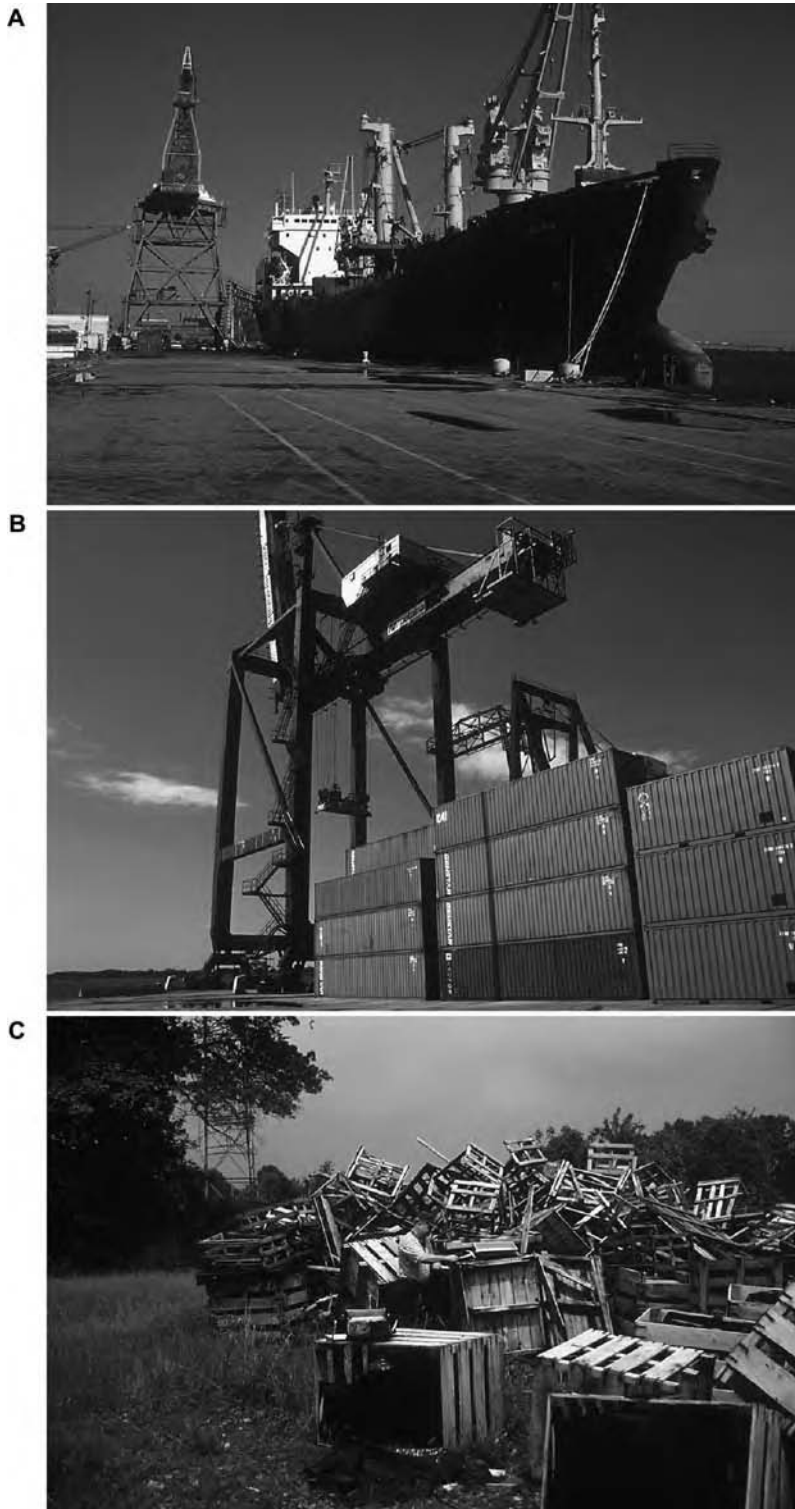


Fig. 3 (A) A container ship moored at the port of Philadelphia, PA. (B) Bulk freight containers being offloaded and stacked at the port of Philadelphia, PA. (C) Wooden packing crates discarded in refuse pile outside an industrial warehouse in Pennsylvania. (Photos by E. R. Hoebeke.)

FUTURE DIRECTION

To safeguard North American agriculture and forestry, it is the principal mandate of the APHIS-PPQ to prevent the entry and the establishment of invasive plant pests in the form of insects, plant diseases, noxious weeds, and

other injurious organisms.^[10] Quarantine and inspection at U.S. ports are the first lines of defense against exotic plant and animal pests and diseases. Because of the recent discovery of an apparent establishment of the Asian long-horned beetle in the United States, USDA-APHIS port personnel will continue to inspect high-risk

cargoes from China for this and other serious wood-boring pests. Federal, state, and city regulatory agencies in New York and Illinois are now committed to the ultimate elimination of the Asian long-horned beetle from North American urban and native landscapes. To this end, a new strategic plan—whose primary objective is to protect the forest products industry, the biological diversity of our hardwood forests and park lands, and the quality of the urban environment from the destructive effects of the Asian long-horned beetle through its containment and elimination—has been implemented that calls for the complete eradication of the Asian long-horned beetle from Chicago by 2008 and from New York by 2009, at a projected cost of US\$86.34 million and US\$254.48 million, respectively.^[11]

REFERENCES

1. U.S. Department of Agriculture, Animal and Plant Health Inspection Service. *Wanted: The Asian Longhorned Beetle*; 1999, Program Aid No. 1655, 10 pp.
2. Mattson, W.J.; Niemela, P.; Millers, I.; Inguanzo, Y. *Immigrant Phytophagous Insects on Woody Plants in the United States and Canada: An Annotated List*; General Technical Report NC-169; USDA Forest Service, North Central Forest Experiment Station, 1994; 27 pp.
3. U.S. Department of Agriculture, Animal and Plant Health Inspection Service and Forest Service. *Pest Risk Assessment for Importation of Solid Wood Packing Materials into the United States*; USDA, APHIS and Forest Service, 2000.
4. U.S. General Accounting Office. Report to Congressional Committees. *Agricultural Inspection: Improvements Needed to Minimize Threat of Foreign Pests and Diseases*; May 1997. GAO/RCED-97-102.
5. Berenbaum, M.R. Two horns, six legs & one voracious appetite. *Audubon Mag.* **2000**, *102* (1), 74–79.
6. Haack, R.A.; Law, K.R.; Mastro, V.C.; Ossenbruggen, H.S.; Raimo, B.J. New York's battle with the Asian long-horned beetle. *J. For.* **1997**, *95* (12), 11–15.
7. <http://www.na.fs.fed.us/spfo/alb/index.htm> (accessed December 2001).
8. <http://www.entomology.wisc.edu/mbcn/fea606.html> (accessed December 2001).
9. Nowak, D.J.; Pasek, J.E.; Sequeira, R.A.; Crane, D.E.; Mastro, V.C. Potential effect of *Anoplophora glabripennis* (Coleoptera: Cerambycidae) on urban trees in the United States. *J. Econ. Entomol.* **2001**, *94* (1), 116–122.
10. National Plant Board. *Safeguarding American Plant Resources: A Stakeholder Review of the APHIS-PPQ Safeguarding System*; U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine: Washington, DC, 1999; 132 pp+ appendices.
11. Stefan, M.; Markham, C. *Strategic Plan for Eradication of Asian Longhorned Beetle from New York and Illinois*; U.S. Department of Agriculture: Riverdale, MD, 2000. An unpublished report, 17 pp. + appendices.

Bacterial Pest Controls

David N. Ferro

Department of Entomology, University of Massachusetts, Amherst, Massachusetts, U.S.A.

INTRODUCTION

Although many genera of bacteria are found to be associated with insects—such as *Clostridium*, *Strategus*, *Pseudomonas*, *Proteus*, *Diplococcus*, *Serratia*, *Bacillus*, and *Enterobacter*—only *Bacillus* and *Serratia* represent agents that cause suppression of insect populations, i.e., that perform as biological control agents. Bacteria are the most widely used microbial agents for controlling insect pests. Some species of *Bacillus* and *Serratia* kill by replicating within the host, while strains of *Bacillus thuringiensis* produce protein toxins that kill soon after being ingested. Bacteria that replicate within their hosts and that persist in the environment by maintaining an infection cycle are biological control agents in the traditional sense. However, products of *B. thuringiensis* that produce toxins that kill insect pests and are applied the way an insecticide would be applied are often not considered to be biological control agents. Bacteria in the genera *Photorhabdus* and *Xenorhabdus* are symbiotic with nematodes in the families Heterorhabditidae and Steinernematidae, respectively. The nematodes serve as vectors that mechanically penetrate into the insect hemocoel and deposit the bacteria. The bacteria then replicate and kill the host quickly by causing septicemia. The only commercially available bacterial products are from strains of *B. popilliae*, *B. thuringiensis*, and *Serratia entomophila*.

PAENIBACILLUS (FORMERLY BACILLUS) POPILLIAE (DUTKY)

Milky disease was first observed in Japanese beetle larvae (grubs) in New Jersey in 1933. *P. popilliae* is an obligate pathogen of larvae in the family Scarabaeidae, as it is only found associated with its host or in the soil surrounding its host. *P. popilliae* and *Paenibacillus lentimorbus* (Dutky) both cause milky disease of scarab beetles; however, most discussions of milky disease refer to strains of *P. popilliae*. *P. popilliae* produces a crystal or parasporal body, which allows it to survive for many years in the soil in the absence of its host. Although there are dozens of strains of *P. popilliae* that infect scarab hosts, only *P. popilliae* has been

used commercially as a biological control agent of the Japanese beetle, *Popillia japonica* (Newman), a major pest of turf.

The term “milky disease” describes the advanced stages of infection in scarab larvae where the host is turned a milky white by the build-up of *Bacillus* spores in the hemolymph. The infection process begins with the scarab larvae ingesting spores while feeding on roots and organic matter in the soil. The spores then undergo germination and outgrowth in the cells of the lumen of the alimentary canal. The vegetative rods penetrate the epithelial cells of the midgut, and then move into the hemolymph where they multiply and sporulate. Death often occurs a month or more after ingestion. It is unclear what the role of the proteinaceous parasporal body is in the infection process.

Culture and Control

Many attempts have been made to rear *P. popilliae* on an artificial diet. Even though spores and vegetative rods from field-collected larvae can be plated on agar media, the inability of the milky disease bacteria to grow and sporulate on standard microbiological media has made it extremely costly to produce for commercial purposes. Products, to date, are made from milky larvae, primarily from naturally infected larvae collected from the field.

The spores are formulated on talc and contain 10^8 spores/g of powder. The powder is applied at about 20 kg/ha using a fertilizer spreader or by punching holes in the soil and adding bacteria. Infection can occur in all three larval stages. For optimal replication to occur, soil temperatures need to exceed 20°C. Large overwintered larvae usually pupate before soil temperatures are high enough in late spring. For this reason, applications are targeted against small larvae late in the summer when the small larvae are actively feeding near the soil surface. Control seems to be greatest when larval densities exceed 300/m²; however, economic losses in turf occur at densities above 100/m². Unless a more virulent strain is found or a more cost-effective way to produce spores is developed, the use of this bacterium is likely to be restricted to lawns and playing fields that can tolerate higher densities of larvae.

SERRATIA ENTOMOPHILA (GRIMONT ET AL.)

Amber disease of the New Zealand grass grub *Costelytra zealandica* (White) is a chronic infection of the larval gut caused by *S. entomophila*. This disease was first observed in New Zealand in 1981. Following ingestion of bacterial cells while feeding on grass roots, the bacteria adhere to the foregut and multiply in the region of the cardiac valve; the larvae cease feeding after 2–5 days and become amber colored due to clearance of the gut. Death does not occur until 1–3 mo after ingestion. As the disease progresses, the larvae become shrunken due to a general degradation of the fat cells. Invasion of the hemocoel does not occur until late stages of the disease, when general septicemia is accompanied by death of the insect.

Culture and Control

S. entomophila is produced in large fermentors as nonspore-forming bacteria to be applied as a live microbial pesticide. Recently, the Industrial Processing Division of DSIR, New Zealand produced 4×10^{10} bacteria/ml, and field trials have shown that $>4 \times 10^{13}$ /ha are needed for control. The problem with using live bacteria (vs. spores) is the difficulty of maintaining viability on the shelf and in the field prior to ingestion. Currently, refrigerated product can be kept for only 3 mo.

Grass grub larvae live in the soil as pests of low-value grasslands. Because *S. entomophila* is applied as live bacteria rather than as spores, it is more vulnerable to UV light and desiccation. For this reason, it is important to place the formulated material 2–5 cm below the soil surface using a subsurface applicator, such as a modified seed drill. This approach allows for 90% survival of the bacteria. Bacteria applied in this way quickly start an epizootic, which then spreads through the grass grub population.

BACILLUS THURINGIENSIS (BERLINER)

B. thuringiensis is a spore-forming bacterium that produces a parasporal crystal (protein delta-endotoxin). After the susceptible insect larva ingests the endotoxin, in the absence or presence of the spore, the crystal is solubilized and activated by alkaline (pH 10.5) gut proteases. The toxic subunits bind to receptor sites on the midgut epithelium within minutes of ingestion. This is quickly followed by lysis of these cells, causing a cessation of feeding within 10–15 min of ingestion. Although the spores pass into the hemocoel through pores in the epithelium of the midgut, it is the starvation in conjunction with infection that kills the

insect. The toxins from these bacteria are formulated in much the same way as a synthetic toxin, and do not cause an epizootic.

There are several subspecies (=strains) of *B. thuringiensis* based on the serotype of flagellar antigens, and these subspecies produce different endotoxins, or at least different amounts of endotoxins that are relatively host specific. For example, *B.t. israelensis* is effective against Nematocera (Diptera) larvae such as mosquito larvae, *B.t. kurstaki* against Lepidoptera, *B.t. aizawai* against Lepidoptera, and *B.t. tenebrionis* against Chrysomelidae (Coleoptera). Notation for the gene that encodes for the toxin is in lowercase; for example, Cry3A gene regulates the production of the Cry3A toxin. Table 1 includes a list of some of the subspecies and toxins they produce. Because these bacteria are so host-specific, they can be quickly incorporated into a pest management program in which biological control agents are an integral component.

Culture and Control

B. thuringiensis can be produced in large quantities using commercial fermentors. Formulations can be applied to foliage or other larval substrates in the same manner as most insecticides. However, several operative factors affect the effectiveness of these bacterial agents.

B.t.s are most effective against early instars (Table 2). Their effectiveness is very dependent upon ambient temperatures; the protein endotoxin is not very persistent; thorough coverage of foliage is

Table 1 *B. thuringiensis* subspecies and crystal protein toxins

| Crystal protein | <i>B.t. subspecies</i> | | | |
|-----------------|------------------------|----------------------|-------------------------|-------------------------|
| | <i>B.t. aizawai</i> | <i>B.t. kurstaki</i> | <i>B.t. tenebrionis</i> | <i>B.t. israelensis</i> |
| Cry1Aa | * | * | | |
| Cry1Ab | * | * | | |
| Cry1Ac | | * | | |
| Cry1C | * | | | |
| Cry1D | * | | | |
| Cry2A | | * | | |
| Cry2B | | * | | |
| Cry3A | | | * | * |
| Cry4A | | | | * |
| Cry4B | | | | * |
| Cry4C | | | | * |
| Cry4D | | | | * |
| CytA | | | | * |

Table 2 Stage-specific larval mortality for the Colorado potato beetle fed foliage treated with *B. thuringiensis san diego* (=tenebrionis)

| Larval stage | LC ₅₀ (mg/l) | Larval weight (mg) | 95% CI | |
|------------------|-------------------------|--------------------|--------|-------|
| | | | Lower | Upper |
| Early 1st instar | 2.03 | 1.0 | 1.46 | 2.60 |
| Late 1st instar | 3.92 | 2.3 | 2.02 | 6.27 |
| Early 2nd instar | 4.35 | 4.0 | 3.30 | 5.56 |
| Late 2nd instar | 14.45 | 7.8 | 10.75 | 19.50 |
| Early 3rd instar | 14.86 | 15.6 | 9.95 | 20.48 |

necessary; and they are host-specific. This host specificity allows for control of the target pest without killing other insect biological control agents; however, in many cropping systems, there is a complex of insect pests and often these need to be controlled at the same time, which may require using the *B.t.* product with a synthetic insecticide, if natural controls fail. Novel ways have been developed to deliver the toxin for ingestion by the pest.

One of the genes that control the production of the toxin has been inserted into *Pseudomonas fluorescens*. After the fermentation has been completed, the broth is chemically treated and heated to kill the bacteria. During this process, the protein toxin becomes encapsulated by the bacterial cell wall. The encapsulation process appears to protect the toxin from degradation in the field, making it more persistent. Several genes have also been inserted into plants that express the toxin in its tissues. In the case of potatoes, the transgenic plants are highly resistant to the Colorado potato beetle, which has considerably reduced the insecticide load on potatoes.

POTENTIAL BIOLOGICAL CONTROL AGENTS

Bacillus sphaericus (Neide) has been shown to be toxic only to larvae of culicid Diptera mosquitoes.

This bacterium can be easily produced via fermentation. Insecticidal activity is due to crystalline toxins associated with the cell wall. The toxin is released by digestion after the host insect has consumed the bacteria. *B. alvei* and *B. brevis* are infectious for larvae of several mosquito species. There is no evidence that these species are significant biocontrol agents. The success of these bacteria in the field is likely to be dependent on selection of strains that are more virulent and that can persist in a range of aquatic environments.

BIBLIOGRAPHY

- Crickmore, N.; Zeigler, D.R.; Feitelson, J.; Schnepf, E.; Van Rie, J.; Lereclus, D.; Baum, J.; Dean, D.H. Revision of the nomenclature for the *Bacillus thuringiensis* pesticidal crystal proteins. *Am. Soc. Micro.* **1998**, *62*, 807–813.
- Glare, T.R.; Jackson, R.A. *Use of Pathogens in Scarab Pest Management*; Intercept: Andover, England, 1992; 43–61, 179–198.
- Jackson, T.A.; Huger, A.M.; Glare, T.R. Pathology of amber disease in the New Zealand grass grub *Costelytra zealandica* (Coleoptera: Scarabaeidae). *J. Invert. Pathol.* **1993**, *61*, 123–130.
- Tanada, Y.; Kaya, H.K. *Insect Pathology*; Academic Press: New York, 1993; 83–146.
- Van Driesche, R.G.; Bellows, T.S., Jr. *Biological Control*; Chapman & Hall: New York, 1996.

Biocontrol: Limits to Use

Joseph D. Cornell

College of Environmental Science and Forestry, State University of New York,
Syracuse, New York, U.S.A.

INTRODUCTION

One often-cited alternative to the use of chemical pesticides is the use of biologically based technologies (BBTs), which are collectively known as “biocontrol.” In contrast to chemical pesticides, biocontrol methods are thought to be highly specific, affecting only one or a narrowly defined class of organisms. Biocontrol methods are therefore thought to present a greatly reduced risk to the environment and to human health.^[1,2] Furthermore, in some situations such as the control of invasive species in wilderness areas, biocontrol is considered to be the only economical method of pest control.^[3] Consequently, biocontrol represents one of the fastest-growing sectors of the U.S. pest control market. As of 1995, the use of all biocontrol methods in the United States accounted for only a small percentage (2–3%) of the total pest control market; however, this percentage is rapidly changing.^[3] Mounting evidence indicates that commonly used biocontrol techniques and organisms share some of the same risks associated with chemical pesticides. Biocontrol agents have been implicated in harmful effects ranging from the local extinction of non-target species to the alteration of entire ecosystems. This suggests that there are limits to the safe use of biocontrol techniques, which must be addressed if we are to continue using them.^[4–6]

There are two main forms of biocontrol, and each carries their own particular risks. The more common form of biocontrol is the use of other organisms that are the “natural enemies” of pest species.^[3] The second, and more recently developed, form of biocontrol is based on direct manipulation of the biology of pest species or their hosts. This includes the use of biochemicals or genetic manipulation, primarily to disrupt the pest’s life cycle, and plant immunization, which seeks to enhance a plant’s resistance to pests through inoculation with chemicals or microorganisms. In addition, combinations of these techniques are emerging; the most notable of which is the insertion of genes from a commonly used biocontrol organism such as *Bacillus thuringiensis* (Bt) into potential host organisms. This technique combines aspects of classical biocontrol with genetic manipulation and plant

inoculation, and the resulting organism represents a new, non-native species that is deliberately introduced into the environment.^[7]

RISKS ASSOCIATED WITH BIOCONTROL

The greatest risks currently associated with biocontrol methods concern the effects of biocontrol agents on native organisms and the ecosystems they inhabit. Biocontrol organisms can directly harm non-pest species through increased predation, parasitism, and herbivory of non-target species; competition with native species; elimination of keystone species; and indirectly through community and ecosystem effects. The next greatest risk concerns the spread of biocontrol organisms and their potential to become pests themselves. In addition, there are risks involved with the release of biologically active chemicals and genetically modified organisms into the environment. Lastly, there is evidence of risks to human health associated with biocontrol methods.

RISK TO NON-TARGET SPECIES

Early attempts at biocontrol using predatory species often resulted in unintended harmful effects in non-target species. One example is the introduction of the Indian Mongoose (*Herpestes auropunctatus*) to control rats on the islands of the West Indies, Hawaii, Mauritius, and Fiji, which resulted in documented declines of native bird species^[4,8] as well as some reptile species in the West Indies.^[9] Other examples include the effects on populations of native birds following the introduction of the barn owl (*Tylo alba*) into Hawaii in 1958 to control rodents, and the extinction of endemic snail species following the introduction of the predatory snail *Euglandina rosea* into island ecosystems throughout the world to control invasions of the giant African snail *Achatina fulica*.^[3] The main problem with all these introductions was that the organisms chosen for biocontrol were “generalists” capable of affecting a wide variety of organisms including non-target species. In each case, the possible

effects on non-target organisms were not adequately determined before the biocontrol agent was released.

Subsequent to these failures, organisms chosen for biocontrol have been more closely screened for specificity in order to reduce the likelihood of directly harming non-target species. Even so, biocontrol organisms are still proving capable of direct harm to non-target species,^[4] especially if the pest and non-target species are closely related.^[5] An example is the introduction of the South American moth *Cactoblastis cactorum* to several Caribbean islands in 1957, 1962, and 1970 to control highly invasive weed species of *Opuntia* cacti for which chemical pesticides and other forms of control were either ineffective or undesirable.^[10] Previously, *Cactoblastis* had been used with great success to control introduced species of cacti in Australia with no discernable adverse effects, largely because Australia does not have any native species of cacti. Following its introduction into the Caribbean, however, *Cactoblastis* caused widespread damage to native, non-weed species of *Opuntia*. Worse, *Cactoblastis* dispersed on its own and became established on other islands; eventually, it reached the United States, where it was responsible for destroying the last few remaining stands of the semaphore cactus (*Opuntia spinosissima*) in the Florida Keys.

A second example is that of the European fly (*Compsilura concinnata*), first introduced in the United States in 1906 to control the gypsy moth (*Lymantria dispar*) and 12 other pest species.^[11] Closely following its introduction, anecdotal evidence began to accumulate suggesting that *Compsilura* was also affecting populations of native silk moth species. However, this has been conclusively demonstrated only recently. Elkington and Boettner^[12] have shown that the European fly is capable of laying its eggs in at least 180 species of insects and has contributed to the decline of silk moth populations in the northeast United States.

THE RISK OF BIOINVASION

The same characteristics that make natural enemies effective (their ability to harm other organisms, to survive, to disperse, and to adapt to new environments) also make them potentially harmful invaders.^[3] Good examples include the grass carp (*Ctenopharyngodon idella*) and the mosquito fish (*Gambusia affinis*) introduced throughout the world to control aquatic weeds and mosquito larvae, respectively. Both fish are now free living throughout much of the United States and are responsible for a variety of harmful effects, including the alteration of aquatic vegetation used by other species (grass carp) and declines in native fish populations as a result of increased predation (mosquito fish).^[3]

RISKS ASSOCIATED WITH GENETIC MODIFICATION

There are two types of risk associated with biocontrol methods based upon genetic modification (GM): the potential transfer of genes from genetically modified individuals into free-living populations of both target and non-target species, and secondly, the potential effects of genetically modified organisms on non-target species and on ecosystems. Genes for Bt corn have recently been found in isolated fields of native criollo maize in Mexico.^[13] Gene transfer from GM corn pollen to the native corn is currently thought to be responsible. In turn, the transfer of genes coding for Bt toxins may exacerbate the harmful effects of Bt toxins on non-target species of plants and insects. Studies have demonstrated harmful effects on non-target species of Lepidoptera as a result of exposure to Bt corn.^[3] Other studies have tried to simulate how increased applications of chemical herbicides made possible by crop plants modified to tolerate heavier doses of herbicides could eliminate food sources for birds.^[14]

THE RISK FROM BIOCHEMICALS

Traps baited with synthetic pheromones to attract pests may also attract coevolved natural enemies, which use the pheromones to find their prey. Chemicals and microorganisms used to inoculate plants against infection can also deter the establishment of beneficial microorganisms and may harm pollinators.

HUMAN HEALTH RISKS

Exposure risks include inhalation of microbial pesticides during application, exposure to wasps and nematodes in schools and kitchens where these biocontrol agents are used, ingestion of microbial pesticides on food, and occupational exposure in rearing facilities.^[3] Documented adverse health effects include allergic reactions to fungal pathogens, and allergic reactions and rhinoconjunctivitis upon exposure to eggs, scales, and wastes of arthropod pests and natural enemies. Additional risks come from the potential contamination of microbial pesticides with human pathogens such as *Shigella* and *Salmonella*.^[15]

MITIGATING THE RISK

The best way to mitigate risk from biocontrol is to rigorously screen potential biocontrol organisms and methods before application.^[5,6] Indeed, because of the ability of many biocontrol organisms to survive,

reproduce, and disperse on their own, prevention may be the only option for reducing risk. Unfortunately, the ability of research to elucidate a priori all possible responses to the introduction of particular biocontrol measures, including ecological effects, effects on non-target species, and human health risks, is limited. For example, microbial pesticides often contain a mixture of organisms in order to attack a broader range of pests than would be possible using a single organism.^[3] Testing for all the possible interactions and ecosystems effects, let alone the risk to human health of these microbial mixtures, simply may not be possible prior to use. Instead, many effects will only become apparent long after the release or introduction of future biocontrol measures. Furthermore, as the effects of biocontrol agents propagate through an ecosystem, it may be difficult to discern the role of biocontrol agents in the decline or disappearance of other organisms.

One example of the extended ecological consequences of using biocontrol concerns the extinction of the large blue butterfly, *Maculina arion*, in England as a result of the introduction of *Myxoma* virus to control rabbit populations.^[16] Reductions in local rabbit populations because of *Myxoma* led to increased growth of vegetation and declines in populations of *Myrmica sabuleti*, an ant which will not build its underground nests in overgrown areas. Finally, the loss of the ant led to the extinction of the blue butterfly, which needs the nests of the ant for its caterpillars to develop into adults. In the words of Simberloff and Stiling,^[4] “What is really remarkable about this convoluted chain of events is not that it occurred, but that anyone noticed it.” More recently, *Myxoma* virus has been used with great success to control introduced rabbits in Australia where it seems to have caused no discernible harmful effects, again because rabbits are a wholly alien species and are not part of Australia’s natural environment.

UNLOCKING PANDORA’S BOX

Ultimately, the continued growth of the human race will necessitate tremendous increases in the extent and intensity of agriculture.^[17,18] In order to avoid the harmful effects of the increased use of chemical pesticides, alternative pest control measures, largely based on biocontrol techniques, will become increasingly important at even faster rates. As such, the human race is placing a great deal of hope on the future capacity of biocontrol to replace chemical pesticides and to do so with fewer harmful effects. But this hope may be yet another Pandora’s box in our attempts to control pests.^[19] Risk from biocontrol methods, as well as most other methods of pest

control, depends on four factors: The range of organisms affected, the reversibility of those effects, the potential for dispersal, and finally, the capacity to monitor and mitigate harmful effects.

CONCLUSION

In general, organisms used for biocontrol do seem to have a much more narrow range of action than most chemical pesticides. Often, however, closely related non-target organisms may be at risk. In regard to reversibility and dispersal, biocontrol agents may actually be worse than chemical pesticides. Chemical pesticides do not have the ability to reproduce themselves or disperse on their own. Indeed, in the case of classical biocontrol, it is usually hoped that introduced natural enemies will permanently establish themselves in order to provide long-lasting relief from pest organisms. Even in the absence of noticeable adverse effects, these introductions represent the beginning of long-term changes to ecosystems and species assemblages, which, in turn, have the potential to significantly alter ecosystem processes.^[18] Many of our current problems with biocontrol can be traced to introductions made many years earlier.^[6] Compounding a lack of adequate testing for the ecological/environmental effects of releasing potential biocontrol agents or technologies, almost no thought is currently given to the potential effects of the alternatives, namely the continued use of chemical pesticides or even doing nothing. In many cases, it is assumed that the use of biocontrol is preferable to the adverse effects of chemical pesticides and to the potential losses incurred by doing nothing. Undoubtedly, this is true in many cases. But without an actual ecological assessment of all three options, no scientific basis exists for evaluating the tradeoffs between alternative strategies.^[4,20]

Lastly, the ability of the United States or any other country to adequately monitor, let alone mitigate, the long-term ecological/environmental effects of biocontrol is questionable. Instead, as the spread of biocontrol occurs, it is highly probable that we will be confronted again and again with the limitations to these techniques until we have examples of nearly every problem now associated with chemical pesticides.

REFERENCES

1. Pimentel, D.; Acquay, H.; Biltonen, M.; Rice, P.; Silva, M.; Nelson, J.; Lipner, V.; Giordano, S.; Horowitz, A.; D’Amore, M. Environmental and economic costs of pesticide use. *BioScience* **1992**, *42* (10), 750–760.
2. Strong, D.R. Fear no weevil? *Science* **1997**, *277*, 1058–1059.

3. OTA. *Biologically Based Technologies for Pest Control*, OTA-ENV-636; U.S. Congress Office of Technology Assessment: U.S. Government Printing Office, Washington, DC, 1995; 204 pp.
4. Simberloff, D.; Stiling, P. How risky is biological control? *Ecology* **1996**, *77* (7), 1965–1974.
5. Pemberton, R.W. Predictable risk to native plants in weed biological control. *Oecologia* **2000**, *125*, 389–494.
6. Strong, D.R.; Pemberton, R.W. Biological control of invading species—risk and reform. *Science* **2000**, *288*, 1969–1970.
7. Ewel, J.J.; O’Dowd, D.J.; Bergelson, J.; Daehler, C.C.; D’Antonio, C.M.; Gómez, L.D.; Gordon, D.R.; Hobbs, R.J.; Holt, A.; Hopper, K.R.; Hughes, C.E.; LaHart, M.; Leaker, R.R.B.; Lee, W.G.; Loope, L.L.; Lorence, D.H.; Louda, S.M.; Lugo, A.E.; McEvoy, P.B.; Richardson, D.M.; Vitousek, P.M. Deliberate introductions of species: research needs. *Bioscience* **1999**, *49* (8), 619–630.
8. Cheke, A.S. An ecological history of the Mascarene Islands, with particular reference to extinctions and introductions of land vertebrates. In *Studies of Mascarene Island Birds*; Diamond, A.W., Ed.; Cambridge University Press: Cambridge, UK, 1987; 5–89.
9. Honegger, R.E. List of amphibians and reptiles either known or thought to have become extinct since 1600. *Biol. Conserv.* **1981**, *19*, 141–158.
10. Johnson, D.M.; Stiling, P.D. Host specificity of *Cactoblastis cactorum* (Lepidoptera: Pyralidae), an exotic *Opuntia*-feeding moth, in Florida. *Environ. Entomol.* **1996**, *743*.
11. Sanchez, V. The Genetic Structure of Northeastern Populations of the Tachinid *Compsilura concinnata* (Meigen), an Introduced Parasitoid of Exotic Forest Defoliators of North America. Ph.D. Thesis, University of Massachusetts: Amherst, 1996.
12. Boettner, G.H.; Elkington, J.S.; Boettner, C.J. Effects of biological introduction on here non-target native species of Saturniid moths. *Conserv. Biol.* **2000**, *14* (6), 1798–1806.
13. Quist, D.; Chapela, I.H. Transgenic DNA introgressed into traditional maize landraces in Oaxaca, Mexico. *Nature* **2000**, *441*, 541–543.
14. Watkinson, A.R.; Freckleton, R.P.; Robinson, R.A.; Sutherland, W.J. Predictions of biodiversity response to genetically modified herbicide-tolerant crops. *Science* **2000**, *289*, 1554–1557.
15. Sjoblad, R.D.; McClintock, J.T.; Engler, R. Toxicological considerations for protein components of biological pesticide products. *Regul. Toxicol. Pharmacol.* **1991**, *15*, 3–9.
16. Ratcliffe, D. The end of the large blue butterfly. *New Sci.* **1979**, *8*, 457–458.
17. Tinker, P.B. The environmental implications of intensified land use in developing countries. *Philos. Trans. R. Soc. Lond., B* **1997**, *352*, 1023–1033.
18. Tilman, D.; Fargione, J.; Wolff, B.; D’Antonio, C.D.; Dobson, A.; Howarth, R.; Schindler, D.; Schlessinger, W.H.; Simberloff, D.; Swackhamer, D. Forecasting agriculturally driven global environmental change. *Science* **2001**, *292*, 281–284.
19. Howarth, F.G. Classical biological control: Panacea or Pandora’s Box? *Proc. Hawaii. Entomol. Soc.* **1983**, *24*, 239–244.
20. Pimentel, D. Biological control of invading species. *Science* **2000**, *289*, 869.

Bioindicators: Use for Assessing Sustainability of Farming Practices

Joji Muramoto

Center for Agroecology and Sustainable Food Systems, University of California-Santa Cruz, Santa Cruz, California, U.S.A.

Stephen R. Gliessman

Department of Environmental Studies, University of California-Santa Cruz, Santa Cruz, California, U.S.A.

INTRODUCTION

The growth of larger-scale monocultures, which heavily depends on the use of pesticides, chemical fertilizers, and agricultural machinery significantly increased crop yield. However, it also brought about negative impacts on ecological (e.g., soil erosion, nitrate, and pesticide contamination of groundwater and loss of agrobiodiversity), economic (e.g., increase of production and marketing costs and decrease of the net income of farmers), and social (e.g., loss of agricultural communities because of farm consolidations) sustainability of agriculture. Since the 1970s and more so 1980s, therefore, various alternative farming practices have been developed in pursuit of sustainable agriculture. Farmers around the world have adopted these practices to varying degrees, but evaluation of their success needs to be conducted locally. Managers and policy makers need tools for assessing changes in agroecosystems to implement sustainable agricultural policies. Good indicators must be simple enough so as to make a very complex framework operate as diagnostic or decision support tools. Bioindicators are important, because they are direct measures of the desired outcome, i.e., increased biodiversity.

This article will first briefly review some concepts and background information regarding bioindicators. Then we discuss the current status of developing bioindicators with two case studies: Europe and Latin America (shaded-coffee systems). The former case represents temperate agroecosystems with 4000 years of farming history. The latter case characterizes upland tropical agroecosystems known to occur in regions of the world with some of the highest, yet most threatened, biodiversity. Further an example of farmer and consumer education for implementing agrobiodiversity policy in Japan is presented.

AGRICULTURAL SUSTAINABILITY AND AGROBIODIVERSITY

Although many definitions of sustainable agriculture exist, all include ecological, economic, and social goals. Sustainable farming practices: (i) maintain their natural resource base; (ii) rely on minimum artificial inputs from outside the farm system; (iii) manage pests and diseases through internal regulating mechanisms; and (iv) recover from the disturbances caused by cultivation and harvest.

Functions of biodiversity in agroecosystems or agrobiodiversity are one of the foundations of sustainable farming practices.^[1] Examples of farming practices that can enhance agrobiodiversity and agroecosystem sustainability are listed in Table 1.

BIOINDICATORS

In the 1980s, bioindicators (e.g., the dieback of lichens) were first applied to detect environmental pollution caused by heavy metals and air pollutants. As biodiversity became the focal point of global and local policies in the 1990s, greater use of bioindicators shifted to evaluation of the status of ecosystems and sustainability of agroecosystems.^[2,3]

Inventory and classification are the foundations for developing indicators.^[4] Developing an indicator then involves several steps: define objectives; determine end-uses; construct indicators; determine norms and thresholds; and testing sensitivity, probability, and usefulness.^[5] Compared to abiotic indicators, however, developing a bioindicator has greater challenges for several reasons. First, biotic parameters are highly variable both temporally and spatially. Information on temporal and spatial variability of most natural species populations, however, is unknown in many places. Secondly, owing to high variability, it costs more to

Table 1 Farming practices that can enhance agrobiodiversity and agroecosystem sustainability

| | |
|--|------------------|
| <i>Habitat diversification</i> | |
| Spatial | |
| | Intercropping |
| | Trap crops |
| | Hedgerows |
| | Shelterbelts |
| | Windbreaks |
| | Agroforestry |
| | Mosaic landscape |
| Temporal | |
| | Rotations |
| | Fallow |
| | Cover crops |
| <i>Conservation or minimum tillage</i> | |
| <i>Organic amendment applications</i> | |
| Compost, organic mulch | |
| <i>Biological pest management</i> | |
| No or reduced use of pesticides, fungicides, herbicides, and fumigants | |
| Use of beneficial insects | |
| <i>Plant resistance</i> | |

collect data. Standardized sampling methods for bioindicators are also lacking. Lastly, it is more difficult with bioindicators to define background levels, norms, and thresholds.^[3]

Nevertheless demands for bioindicators as evaluation tools at diverse levels are on the rise. Some of these are policy driven, and others are market driven. For example, international organizations, such as Organisation for Economic Co-operation and Development^[6] and European Union (EU),^[7] promote development of standardized bioindicators as a policy to compare biodiversity worldwide. Among commercial sectors, bioindicators have been used to certify “environmentally sound” products, such as migratory birds for shade-grown coffee.^[8]

BIOINDICATOR DEVELOPMENT IN EU AGROECOSYSTEMS^[9]

The importance of agroenvironmental indicators has been highlighted by the EU.^[7] Intensive studies on bioindicators have been conducted in European countries. Bioindicators demonstrated to be sensitive to farm management intensities in European agroecosystems are listed in Table 2. Generally, it is observed among invertebrate species that with less intensive management, there are more specialists and less generalists; greater diversity; and higher abundance.^[3] An example of a bioindicator based on these correlations

is European spiders; habitat preferences of spiders, particularly the ratio of “pioneer species (mostly Linyphiidae)” vs. “wolf spiders (Araneae: Lycosidae),” can be a sensitive indicator for the assessment of farming intensity.^[10] Many bioindicators listed in Table 2, however, have critical use limitations owing to technically complex sampling methods and greater temporal and spatial variability.

To practically implement agroenvironmental policy in the EU, efforts have been made to develop relatively easy-to-measure surrogate indicators (e.g., length of borders, farm size, and area managed with organic farming).^[3] Another practical bioindicator is a list of indicator plant species to evaluate species richness of a farm. Twenty-eight indicator flower species for meadows and pastures, which can be easily identified by local farmers, were selected in Baden-Württemberg, Germany. Agroenvironmental payments are granted to farms that have at least 4 of these 28 indicator species in all of the meadows and pastures on the farm.^[11] It has been further recognized that the preservation of biodiversity is only possible through the (re) establishment of a mosaic of habitat patches at the landscape level. To meet this need, GIS-based landscape-oriented indicators have been examined.^[12]

BIOINDICATOR DEVELOPMENT IN SHADED COFFEE IN LATIN AMERICA

Coffee is the second most traded commodity in the world after petroleum, and forms the principal economic activity of more than 20 million people in farming communities throughout much of the developing world. Traditionally grown in the understory of forest cover or planted shade trees, throughout the 1970s and 1980s, many coffee farmers adopted more modern production practices—planting higher yielding varieties in full sun, eliminating shade trees, and increasing pesticide and fertilizer applications.^[13] The loss of shade cover, and associated biodiversity, has led to many environmental problems such as soil erosion, loss of water capture and recharge ability, and contamination from the excessive fertilizer and pesticide use associated with sun-grown coffee.

But recent research has documented the high levels of agrobiodiversity and ecosystem services (such as pollination, soil conservation, and natural pest control) associated with diverse shade coffee production.^[14] From this research, several very important bioindicators are being developed. The species diversity of ants and insect feeding birds is higher in shade coffee than in sun coffee, especially ground-foraging ants that act as important predators, and birds that are bark gleaners and leaf surface foragers. It also appears that the higher diversity of predaceous ants restricts the development of pest ants

Table 2 Examples of potential bioindicators for sustainability of farming practices in European agroecosystems

| Bioindicator | Parameter | Comments |
|--|--|--|
| <i>Arthropods</i> | | |
| Ground beetles (Carabidae) | Abundance | <ul style="list-style-type: none"> • Sensitive to management intensity but needs intensive data collection |
| Spiders (Araneae) | Habitat preferences Percent pioneer species | <ul style="list-style-type: none"> • Highly sensitive to management intensity, and database is available on ecological characteristics of central European spiders |
| Hoverflies (Syrphidae) | Percent stenotopic species | <ul style="list-style-type: none"> • Diversity of landscape structure adjacent to the field enhances species numbers |
| <i>Soil fauna</i> | | |
| Earthworm | Biomass | <ul style="list-style-type: none"> • Suitable indicator for soil structure or compaction, tillage practice, heavy metals, and pesticides |
| | Species number | |
| | Ecological guilds | |
| Collembola | Physiotype | <ul style="list-style-type: none"> • Highly sensitive to management intensity but time consuming, and special skills are required for identification |
| Protozoa | Biodiversity | |
| Nematode | Trophic index Maturity index | |
| <i>Soil microbiota</i> | | |
| Soil enzymes (e.g., proteases, nitrite reductases) | Activities | <ul style="list-style-type: none"> • Moderately sensitive to management intensity and relatively easy to measure |
| Microbial communities | Composition | <ul style="list-style-type: none"> • Moderately sensitive to management intensity, but special skills are required and difficult to interpret |
| | Functional diversity | |
| Functional groups | Mycorrhizae | <ul style="list-style-type: none"> • Highly sensitive to management intensity, but special skills are required |
| | Nitrification | |
| | Root pathogens | |
| Microbial activity | Soil respiration | <ul style="list-style-type: none"> • Relatively easy to measure but highly variable both temporally and spatially |
| <i>Plants</i> | | |
| Higher plants | Numbers of “characteristic” species, functional groups, and endangered species | <ul style="list-style-type: none"> • Capable of being integrated into sophisticated floristic diversity at the habitat scale but requires intensive data collection |
| | Cover of litter in vegetation | |
| | Evenness indices | |
| | Habitat age | |

(Adapted from Ref.^[9].)

such as the fire ant *Solenopsis* sp. (Hymenoptera), a very common pest in open landscapes and sun-grown coffee. Associated species such as orchids in the shade tree canopy can also be important bioindicators. Local farmers can be trained to recognize orchids, demonstrating how the development of bioindicators of sustainability can be an accessible and local methodology that adds value to more sustainable farming practices. The orchids have an intrinsic value from a conservation perspective, while at the same time in this region of Northern Nicaragua have added an attractive value for an emerging agroecotourism industry associated with shade grown coffee landscapes.^[15]

IMPROVING AWARENESS OF FARMERS AND CONSUMERS

It is not rare that farmers themselves are not fully aware of the biodiversity in their farms. Moreover, to economically sustain environmentally friendly farming practices, recognition and support from consumers are necessary. To implement sustainable agricultural policy, therefore, improved awareness of both farmers and consumers on agrobiodiversity is required.

An example of such educational activity is the biodiversity inventory in paddy rice ecosystems in Japan, where paddy fields occupy ~50% of the cultivated

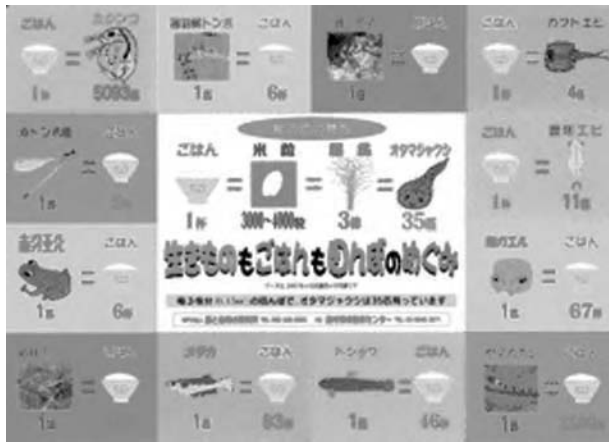


Fig. 1 Part of a Japanese poster for improving farmer and consumer awareness of agrobiodiversity in paddy fields. Central figure indicates a bowl of rice being equivalent to 3000–4000 grains of rice, three stubs of rice plants, and 35 tadpoles in a paddy. This can be interpreted that eating a bowl of rice supports 35 tadpoles in a paddy. Numbers of species are averages from the national survey of agrobiodiversity in paddy fields in Japan. Clockwise from the upper left, species listed are *Daphnia pulex*, *Pantala flavescens*, *Sympetrum frequens*, *Triopus longicaudatus*, *Branchinella kugenumaensis*, *Hyla japonica*, *Rhabdophis tigrinus*, *Misgurnus anguillicaudatus*, *Oryzias latipes*, *Rana nigromaculata*, *Rana rugosa*, and *Ceriatrion melanurum*. (With permission from Ref.^[18].)

lands of the country. Japanese paddy agroecosystems contain a diverse group of organisms, such as birds (60 spp.), fish (70 spp.), reptiles (12 spp.), amphibian (20 spp.), arthropods (>600 spp.), and weeds (190 spp.).^[16] Since the 1960s, however, many native species inhabiting paddy fields have decreased in abundance mainly owing to the side effects of pesticide applications and the construction of concrete ditches. Consequently, two insect species, *Lethocerus deyrollei* (Hemiptera: Lethocerinae) and *Cybister tripunctatus orientalis* (Coleoptera: Dytiscidae), five bird species, one species of fish, and one amphibian, and three plant species are listed as endangered species inhabiting paddy fields.^[17]

NGOs, scientists, farmers, the general public, and administrators collaborate to create nationwide inventories of biodiversity in paddy agroecosystems as the foundation for developing bioindicators, and enhance the awareness of both farmers and consumers on paddy field biodiversity (Fig. 1).^[18]

CONCLUSIONS

Indicators represent a compromise between scientific knowledge of the moment and simplicity of use.^[5] Compared to assessment systems for natural ecosys-

tems,^[4] the current status of developing bioindicators for sustainability of farming practices appears to still be in its infancy. Further, to practice bioindicators to implement sustainable agricultural policy, we need not only more research on the science of bioindicators, but also the better awareness on agrobiodiversity among farmers and consumers.

Future studies on bioindicators for sustainable farming practices should address the following:

1. The importance of stability and reproductive potential of not only pest and beneficial species but also other species typical of agroecosystems.^[3,16]
2. Standardization of sampling methods.
3. Development of multiple sets of bioindicators tailored to different end-users (general public, farmers, policy makers, and scientists).
4. Construction of a hierarchical system that integrates different types of bioindicators.

ACKNOWLEDGMENTS

We thank Drs. Yutaka Une, Keiji Kiritani and Kazumasa Hidaka and an anonymous reviewer for their valuable comments on a previous version of the manuscript.

ARTICLES OF FURTHER INTEREST

- Crop Diversity for Pest Management*, p. 162.
Cultural Control, p. 174.
Ecological Aspects of Pest Management, p. 211.
Hoverflies: Indicators of Sustainable Farming and Potential Control of Aphids, p. 1.
Integrated Farming Systems, p. 410.
Intercropping for Pest Management, p. 423.
Natural Enemy Monitoring in Pest Management, p. 1.
Natural Vegetation Management to Improve Parasitoids in Framing Systems, p. 1.
Nonchemical or Pesticide-Free Farming, p. 533.
Organic Farming, p. 554.
Pest Management in Ecological Agriculture, p. 576.
Pest Population Monitoring, p. 587.
Sustainable Agricultural Practices, p. 812.
Systems Management, p. 826.

REFERENCES

1. Gliessman, S.R. *Agroecology: Ecological Processes in Sustainable Agriculture*; Lewis Publishers: Boca Raton, FL, 1998.

2. Paoletti, M.G. Using bioindicators based on biodiversity to assess landscape sustainability. *Agric. Ecosyst. Environ.* **1999**, *74* (1–3), 1–18.
3. Buchs, W. Biodiversity and agri-environmental indicators-general scopes and skills with special reference to the habitat level. *Agric. Ecosyst. Environ.* **2003**, *98* (1–3), 35–78.
4. Innis, S.A.; Naiman, R.J.; Elliott, S.R. Indicators and assessment methods for measuring the ecological integrity of semi-aquatic terrestrial environments. *Hydrobiologia* **2000**, *422–423*, 111–131.
5. Girardin, P.; Bockstaller, C.; Van der Werf, H. Indicators: tools to evaluate the environmental impacts of farming systems. *J. Sustain. Agric.* **1999**, *13* (4), 5–21.
6. Organisation for Economic Co-operation and Development (OECD). *Environmental indicators for agriculture*; Organisation for Economic Co-operation and Development; OECD Washington Center distributor: Paris, France/Washington, DC, 1997.
7. The Commission of the European Communities. Evaluation of agri-environment programmes. State of application of regulation (EEC) No. 2078/92. DGVI Commission Working Document VI/7655/98. 1998.
8. Mas, A.H.; Dietsch, T.V. Linking shade coffee certification to biodiversity conservation: butterflies and birds in Chiapas. Mexico. *Ecol. Appl.* **2004**, *14* (3), 642–654.
9. Buchs, W., Ed.; Biotic indicators for biodiversity and sustainable agriculture. *Agric. Ecosyst. Environ.* **2003**, *98* (1–3), 1–547 (Special Issue).
10. Buchs, W.; Harenberg, A.; Zimmermann, J.; Wei, B. Biodiversity, the ultimate agri-environmental indicator? Potential and limits for the application of faunistic elements as gradual indicators in agroecosystems. *Agric. Ecosyst. Environ.* **2003**, *98* (1–3), 99–123.
11. Oppermann, R. Nature balance scheme for farms-evaluation of the ecological situation. *Agric. Ecosyst. Environ.* **2003**, *98* (1–3), 463–475.
12. Osinski, E. Operationalisation of a landscape-oriented indicator. *Agric. Ecosyst. Environ.* **2003**, *98* (1–3), 371–386.
13. Moguel, P.; Toledo, V.M. Biodiversity conservation in traditional coffee systems of Mexico. *Conserv Biol* **1999**, *13*, 11–21.
14. Perfecto, I.; Mas, A.; Dietsch, T.; Vandermeer, J. Conservation of biodiversity in coffee agroecosystems: a tri-taxa comparison in southern Mexico. *Biodiversity Conserv* **2003**, *12*, 1239–1252.
15. Bacon, C.M. Confronting the Coffee Crisis: Nicaraguan Farmers' Use of Cooperative, Fair Trade, and Agro-ecological Networks to Negotiate Livelihoods and Sustainability. Ph.D. dissertation, Department of Environmental Studies, University of California, Santa Cruz, CA, USA, 2005.
16. Kiritani, K. Integrated biodiversity management in paddy fields: shift of paradigm from IPM toward IBM. *Integrated Pest Manage. Rev.* **2000**, *5* (3), 175–183.
17. Hidaka, K. Biodiversity conservation and environmentally regenerated farming systems in rice paddy fields. *Jpn. J. Ecol.* **1998**, *48*, 167–178 (in Japanese).
18. Une, Y. *Illustrated Poster of Rice and Living Organisms*; Nou-to-shizen-no-kenkyujo: Fukuoka, Japan, 2004 (in Japanese).

Biological Control of Stored-Product Pests

Lise Stengård Hansen

Danish Pest Infestation Laboratory, Danish Institute of Agricultural Sciences,
Kgs. Lyngby, Denmark

INTRODUCTION

Stored grain and other durables of plant origin, raw and processed, can become infested by insect and mite pests. As the environment in storage facilities in general is conducive to rapid pest development, this often results in substantial quantitative and qualitative losses. Many stored-product pests are internal feeders developing inside whole-grain cereals and legumes in storage. Another group of pests are external feeders developing on broken kernels, flour, etc. and are mainly found in processing facilities, stores of processed cereals and bulk grain, and packaged foodstuffs (Table 1).

Beneficial insects are often found in storage facilities and may prevent or delay pest development. The internal feeders are attacked by a range of parasitoids specialized in detecting infested kernels and placing their progeny on the larvae within the kernel. The external feeders are more freely exposed to the activities of natural enemies and are attacked by both predators and parasitoids (Table 1).

Although the concept is not new, biological control of stored-product pests is still not widely used. The following description gives some examples of the recent development within biological control of stored-product pests using beneficial insects and mites.

For general reviews on stored-product pest species and biological control of stored-product pests, see Refs.^[1-4]

FACTORS THAT PROMOTE SUCCESSFUL BIOCONTROL IN STORAGE

Storage facilities are ideally suited for development and reproduction of pests. However, these factors are also conducive to successful biocontrol: 1) Climatic conditions are relatively stable and, at least during part of the year, favorable for insect development; 2) the storage structure provides protection from temperature extremes and precipitation as well as a physical barrier to dispersal; and 3) in many storage situations, time is not a limiting factor and thus often sufficient for natural enemies to establish and exert their control. These factors can be manipulated to be more favorable

to the natural enemies, e.g., grain temperature can be reduced by aeration to a level that offers a greater advantage to the activities of the beneficial insect than to the pest.^[5]

“CLASSICAL” BIOLOGICAL CONTROL

In “classical” biological control, a natural enemy is imported and released for establishment to control an introduced (exotic) pest. An example of this among stored-product pests is the larger grain borer, *Prostephanus truncatus*, which was accidentally introduced from Central America into Africa, where it has become a serious pest of many products, e.g., stored maize and cassava chips. A predator, *Teretrius nigrescens*, was introduced from Central America and, after large scale releases, established successfully in many locations in Africa. *T. nigrescens* has been credited with reducing losses of stored maize to *P. truncatus*. However, despite the presence of the predator, *P. truncatus* densities often reach outbreak levels in many countries. An analysis in Ref.^[6] led to the conclusion that *T. nigrescens* alone is unable to exert control to an acceptable level inside a store due to the predator’s intraspecific density-dependence and low growth rate compared with its prey. In this case, biocontrol must be supplemented with other integrated control measures.^[7]

BIOCONTROL OF INTERNALLY FEEDING PESTS

The larvae of internally feeding insect pests develop within whole-grain cereals and legumes. These species are important primary pests in stores of bulk grain. Several species of parasitoids are specialized to live on these pests. The adult parasitoids enter the bulk of grain and find infested kernels, probably by means of acoustic or olfactory cues. They then drill into the kernel, paralyze the host, and deposit an egg from which the parasitoid larva emerges. After consuming the host, the parasitoid exits the kernel as an adult. The ability of the parasitoid to locate its host within grain varies among species: *Anisopteromalus calandrae* is primarily active at the grain surface whereas

Table 1 Examples of pests and natural enemies in stored products

| Pest type | Commodity | Examples of pest species | Examples of natural enemies |
|-------------------------------------|---|---|---|
| Internal feeders or primary pests | Whole-grain cereals | <i>Sitophilus</i> spp. (C) <i>Rhyzopertha dominica</i> (C) <i>Sitotroga cerealella</i> (L) <i>Prostephanus truncatus</i> (C) | <i>Lariophagus distinguendus</i> (l) <i>Theocolax elegans</i> (l) <i>Anisopteromalus calandrae</i> (l) <i>Teretrius nigrescens</i> (p) |
| | Whole legumes | <i>Bruchus</i> spp. (C) <i>Callosobruchus</i> spp. (C) | <i>Dinarmus</i> spp. (l) |
| External feeders or secondary pests | Broken kernels, flour, milled rice, dried fruit, spices, nuts | <i>Tribolium</i> spp. (C) <i>Cryptolestes</i> spp. (C) <i>Oryzaephilus</i> spp. (C) | <i>Trichogramma</i> spp. (e) <i>Holepyris silvanidis</i> (l) <i>Cephalonomia</i> spp. (l) <i>Xylocoris flavipes</i> (p) |
| | | <i>Ephestia kuehniella</i> (L) <i>Cadra cautella</i> (L) <i>Plodia interpunctella</i> (L) | <i>Trichogramma</i> spp. (e) <i>Venturia canescens</i> (l) <i>Habrobracon hebetor</i> (l) <i>Xylocoris flavipes</i> (p) |
| | | Storage mites (Acarina) | <i>Cheyletus eruditus</i> (p) |

C: Coleoptera; e: egg parasitoid; L: Lepidoptera; l: larval parasitoid; p: predator. (Based on Refs.^[2-4,12,13])

Theocolax elegans and *Lariophagus distinguendus* are able to find infested kernels down to a depth of 2.2 and 4 m, respectively.^[8,9] Great differences in life history parameters occur among different parasitoid strains.^[8] These factors as well as grain characteristics such as kernel size and grain variety all affect the ability of the natural enemy to exert effective control of the pest.

A constraint against biocontrol in stored products is a reluctance to increase the amount of insects by releasing natural enemies. However, these minute parasitoids (<2 mm) can easily be removed by grain cleaning procedures. In a study using *T. elegans* against *Rhyzopertha dominica* in wheat, the number of insect fragments in the resulting flour was reduced by 89%.^[10]

BIOCONTROL OF EXTERNALLY FEEDING PESTS

Both beetles and moths are represented among external pests that feed on broken kernels and debris as well as flour. They are important pests in grain stores, in cereal processing facilities such as flourmills, and in warehouses storing cereal products.

Many species of parasitoids, particularly Trichogrammatidae, attack the egg stage of external pests. The impact of these species is increased by their host feeding behavior, which can account for half of the mortality of host eggs.^[11] These egg parasitoids are able to parasitize a wide range of species, but in nature they generally show affinity to a specific habitat; it is thus important to select strains that are adapted to

the stored-product environment. *Trichogramma* species have been successfully released against pyralid moth pests in experimental peanut storages in the U.S.A.^[12] and in wholesale stores and industrial bakeries in Germany.^[13] These egg parasitoids do not establish within the premises and must be released on a regular basis.

Larvae of moth pests are attacked by both ectoparasitoids, e.g., *Habrobracon hebetor* and endoparasitoids, e.g., *Venturia canescens*. Both species are cosmopolitan, often occurring together in flourmills and both species show potential for biocontrol of moth pests.^[12] However, *H. hebetor* may affect populations of *V. canescens* negatively by feeding on hosts parasitized by this species.

The predatory bug *Xylocoris flavipes* is a generalist living on eggs and larvae of a wide range of beetles and moths. Almost 30 species of stored-product pests have been reported as prey of *X. flavipes*.^[12] In residues in empty maize stores in the U.S.A., many of these species occur together in the same store. A single introduction of *X. flavipes* led to population reductions of 70–100% of externally feeding beetle pests, whereas internal feeders and late instar moth larvae were less affected. It was suggested that releases of predatory bugs combined with parasitoids for the moths and internal feeders might eliminate or greatly reduce residual pests before the next storage season.^[12]

This strategy of introducing predators in empty stores was widely practiced to control storage mites (Acarina) in the Czech Republic. The predatory mite *Cheyletus eruditus* led to reductions in storage mite populations of 88%, compared with the 18% reduction obtained with a pesticide treatment.^[14]

CONCLUSIONS

The above-mentioned examples show the great potential of biological control of stored-product pests. This research field benefits from extensive international collaboration and applicability, as the pest species as well as their natural enemies have a cosmopolitan distribution as a result of international grain trade. A great amount of faunistic surveys and laboratory research on the natural enemies has been carried out, but very few field trials have been conducted. Application of natural enemies to control pests in bulk grain as well as in empty grain stores prior to introduction of newly harvested grain is considered to hold potential in the near future. However, widespread use depends on crucial experience to be obtained from field trials. The next step is ensuring reliable supplies of natural enemies, designing introduction strategies, and establishing quality control during mass rearing and shipment of beneficials. From these starting points, other pest species and other storage situations can be covered. These activities can contribute to satisfying public demands of more environmentally friendly pest control methods and food production that is focused on consumer safety.

REFERENCES

1. Gorham, J.R., Ed.; *Ecology and Management of Food-Industry Pests*; Food and Drug Administration Technical Bulletin 4; Association of Official Analytical Chemists: Arlington, 1991.
2. Schöller, M. Biologische Bekämpfung vorratschädlicher Arthropoden mit Räubern und Parasitoiden—Samelbericht und Bibliografie (Biological control of arthropod pests in stored product protection with predators and parasitoids—review and bibliography). *Mitt. Biol. Bundesanst. Land- Forstwirtschaft.* **1998**, *342*, 85–189.
3. Subramanyam, B., Hagstrum, D., Eds.; *Integrated Management of Insects in Stored Products*; Marcel Dekker, Inc.: New York, 1996.
4. Subramanyam, B., Hagstrum, D., Eds.; *Alternatives to Pesticides in Stored-Product IPM*; Kluwer Academic Publishers: Boston, 2000.
5. Flinn, P.W. Temperature effects on efficacy of *Choetospila elegans* (Hymenoptera: Pteromalidae) to suppress *Rhyzopertha dominica* (Coleoptera: Bostrichidae) in stored wheat. *J. Econ. Entomol.* **1998**, *91*, 320–323.
6. Holst, N.; Meikle, W.G. *Teretrius nigrescens* against larger grain borer *Prostephanus truncatus* in African maize stores: biological control at work?. *J. Appl. Ecol.* **2003**, *40*, 307–319.
7. Markham, R.; Borgemeister, C.; Meikle, W.G. Can biological control resolve the larger grain borer crisis? In *Stored Product Protection*, Proceedings of the 6th International Working Conference on Stored-product Protection, Canberra, Australia, Apr 17–23, 1994; Highly, E., Wright, E.J., Banks, H.J., Champ, B.R., Eds.; CAB: Wallingford, U.K., 1994; 1087–1097.
8. Steidle, J.L.M.; Schöller, M. Fecundity and ability of the parasitoid *Lariophagus distinguendus* (Hymenoptera: Pteromalidae) to find larvae of the granary weevil *Sitophilus granarius* (Coleoptera: Cucurionidae) in bulk grain. *J. Stored Prod. Res.* **2002**, *38*, 43–53.
9. Press, J.W. Comparative penetration efficacy in wheat between the weevil parasitoids *Anisopteromalus calandrae* and *Choetospila elegans*. *J. Entomol. Sci.* **1992**, *27*, 154–157.
10. Flinn, P.W.; Hagstrum, D.W. Augmentative releases of parasitoid wasps in stored wheat reduces insect fragments in flour. *J. Stored Prod. Res.* **2001**, *37*, 179–186.
11. Hansen, L.S.; Jensen, K.-M.V. Effect of temperature on parasitism and host-feeding of *Trichogramma turkestanica* (Hymenoptera: Trichogrammatidae) on *Ephestia kuehniella* (Lepidoptera: Pyralidae). *J. Econ. Entomol.* **2002**, *95*, 50–56.
12. Brower, J.H.; Smith, L.; Vail, P.V.; Flinn, P.W. Biological control. In *Integrated Management of Insects in Stored Products*; Subramanyam, B., Hagstrum, D., Eds.; Marcel Dekker, Inc.: New York, 1996; 223–286.
13. Schöller, M.; Flinn, P.W. Parasitoids and predators. In *Alternatives to Pesticides in Stored-Product IPM*; Subramanyam, B., Hagstrum, D., Eds.; Kluwer Academic Publishers: Boston, 2000; 229–271.
14. Zdarkova, E. Personal experience with biological control of stored food mites. *IOBC/WPRS Bull.* **1998**, *21* (3), 89–93.

Biotechnology

Maurizio G. Paoletti

Dipartimento di Biologia, Universita di Padova, Padova, Italy

INTRODUCTION

Disease and insect pest resistance to various pests has been slowly bred into crops for the past 13,000 years; current techniques in biotechnology now offer opportunities to further and more rapidly improve the non-chemical control of disease and insect pests of crops. However, relying on a single factor, like the *Bacillus thuringiensis* (BT) toxin that has been inserted into corn, cotton, and a few other crops for insect control, leads to various environmental problems, including insect resistance, and a serious threat to beneficial biological control insects and endangered species. A major environmental and economic cost associated with genetic engineering applications in agriculture relates to the use of herbicide-resistant crops (HRCs). In general, HRC technology results in increased herbicide use, pollution of the environment, and weed control costs for farmers that may be twofold greater than standard weed control costs. Therefore, pest control with both pesticides and genetic engineering methods can be improved for effective, safe, and economical pest control.

BENEFITS OF GENETIC ENGINEERING IN PEST CONTROL

Since 1987, many crops have been genetically modified for features such as resistance to insects, resistance to pathogens (including viruses) and herbicides, and for improved features such as longer-lasting ripening, higher nutritional status, protein content, seedless fruit, and sweetness. Up to 34 new genetically engineered crops have been approved to enter into the market.

In 1998, 27.8 million ha of engineered crops were planted in countries such as the United States, Argentina, Canada, and Australia. The United States alone contains 74% of the modified crop land-planted. Globally, 19.8% of this area has been planted with herbicide-tolerant crops, 7.7% with insect-resistant crops, and 0.3% with insect and HRCs. Five crops—soybean, corn, cotton, canola, and potato—cover the largest acreage of engineered crops.^[1,2]

DISEASE RESISTANCE IN CROPS

The crops currently on the market that have been engineered for resistance to plant pathogens are listed in Table 1. Disease-resistant engineered crops have some potential advantages because few current pesticides can control bacterial and viral diseases of crops. In addition, these engineered plants help reduce problems from pesticides.

The large-scale cultivation of plants expressing viral and bacterial genes might lead to adverse ecological consequences. The most significant risk is the potential for gene transfer of disease resistance from cultivated crops to weed relatives. For example, it has been postulated that a virus-resistant squash could transfer its newly acquired virus-resistant genes to wild squash (*Cucurbita pepo*), which is native to the southern United States. If the virus-resistant genes spread, newly disease-resistant weed squash could become a hardier, more abundant weed. Moreover, because the United States is the origin for squash, changes in the genetic make-up of wild squash could conceivably lessen its value to squash breeders.

Some plant pathologists have also suggested that development of virus-resistant crops could allow viruses to infect new hosts through transencapsidation. This may be especially important for certain viruses, e.g., luteoviruses, where possible heterologous encapsidation of other viral RNAs with the expressed coat protein is known to occur naturally. With other viruses, such as the PRV that infects papaya, the risk of hetero-encapsidation is thought to be minimal because the papaya crop itself is infected by very few viruses.

Virus-resistant crops may also lead to the creation of new viruses through an exchange of genetic material or recombination between RNA virus genomes. Recombination between RNA virus genomes requires infection of the same host cell with two or more viruses. Several authors have pointed out that recombination could also occur in genetically engineered plants expressing viral sequences of infection with a single virus, and that large-scale cultivation of such crops could lead to increased possibilities of combinations. It has recently been shown that RNA transcribed from a transgene can recombine with an infecting virus to produce highly virulent new viruses.

Table 1 Plants genetically-engineered for virus resistance that have been approved for field tests in the United States from 1987 to July 1995

| Crop | Disease(s) | Research organization |
|-----------------------------|--|-----------------------------------|
| Alfalfa | Alfalfa mosaic virus, Tobacco mosaic virus (TMV), Cucumber mosaic virus (CMV) | Pioneer Hi-Bred |
| Barley | Barley yellow dwarf virus (BYDV) | USDA |
| Beets | Beet necrotic yellow vein virus | Betaseed |
| Cantelopes and/or squash | CMV, papaya ringspot virus (PRV) Zucchini yellow mosaic virus (ZYMV), Watermelon mosaic virus II (WMVII) | Upjohn |
| | CMV | Harris Moran Seed |
| | ZYMV | Michigan State University |
| | ZYMV | Rogers NK Seed |
| | Soybean mosaic virus (SMV) | Cornell University |
| | SMV, CMV | New York State Experiment Station |
| Corn | Maize dwarf mosaic virus (MDMV) Maize chlorotic mottle virus (MCMV), Maize chlorotic dwarf virus (MCDV) | Pioneer Hi-Bred |
| | MDMV | Northup King |
| | MDMV | DeKalb |
| | MDMV | Rogers NK Seed |
| Cucumbers | CMV | New York State Experiment Station |
| Lettuce | Tomato spotted wilt virus (TSWV) | Upjohn |
| Papayas | PRV | University of Hawaii |
| Peanuts | TSWV | Agracetec |
| Plum Trees | PRV, plum pox virus | USDA |
| Potatoes | Potato leaf roll virus (PLRV), Potato virus X (PVX), Potato virus Y (PVY) | Monsanto |
| | PLRV, PVY, late blight of potatoes | Frito-Lay |
| | PLRV | Calgene |
| | PLRV, PRY | University of Idaho |
| Potatoes | PLRV, PVY | USDA |
| | PYV | Oregon State University |
| Soybeans | SMV | Pioneer Hi-Bred |
| Tobacco | ALMV, tobacco etch virus (TEV), Tobacco vein mottling virus | |
| | TEV, PVY | University of Florida |
| | TEV, PVY | North Carolina State University |
| | TMV | Oklahoma State University |
| | TEV | USDA |
| Tomatoes | TMV, tomato mosaic virus (TMV) | Monsanto |
| | CMV, tomato yellow leafcurl virus | |
| | TMV, ToMV | Upjohn |
| | ToMV | Rogers NK Seed |
| | CMV | PetoSeed |
| | CMV | Asgrow |
| | CMV | Harris Moran Seeds |
| | CMV | New York State Experiment Station |
| | CMV | USDA |

(From Refs.^[4,5])

A strategy for reduced risk would include: 1) identification of potential hazards; 2) determination of frequency of recombination between homologous, but non-identical sequences in crops and weeds; and 3) determination of whether or not such recombinants can have selective advantage.

ASSESSMENT OF TRANSGENIC VIRUS-RESISTANT POTATOES IN MEXICO

An in-depth assessment of potential socioeconomic implications related to the introduction of some genetically modified varieties of virus-resistant potatoes (PVY, PVX, PIRV) in Mexico underscores the importance of this technology. This type of genetic modification could prove especially beneficial to large-scale farmers, but only marginally beneficial to small-scale farmers, because most small farmers use red potato varieties that are not considered suitable for transformation. In addition, 77% of the seeds that small farmers use come from informal sources, not from the seed providers that could sell the new resistant varieties.

The mycoplasma and virus diseases in Mexico are not currently controlled with pesticides, and rank second and third in economic damages. The major pest, the fungus *Phytophthora infestans*, ranks first in economic damages and requires, in some cases, up to 30 fungicide applications. Thus, the interesting new genetically altered varieties of potatoes are of little benefit to crop production for small farmers.

HRCs

Several engineered crops that include herbicide resistance are commercially available; 13 other key crops in the world are ready for field trials (Table 2). In addition, some crops (e.g., corn) are being engineered to contain both herbicide (glyphosate) and biotic insecticide resistance (BT α -endotoxin).

Herbicides adopted for HRCs employ lower doses when compared with atrazine, 2,4-D, and alachlor. However, the resistance of the crop to the target herbicide would, in practice, suggest to the farmer to apply dosages higher than recommended. In addition, costs for this new technology of HRCs are about two times higher in corn than the recommended herbicide use and cultivation weed control program.

Integrated pest management (IPM) could benefit from some HRCs if alternative non-chemical methods can be applied first to control weeds and the target herbicide could be used later, only when and where the economic threshold of weeds is surpassed. Generally, however, the use of HRCs will lead to increased

use of herbicides and environmental and economic problems. Most HRCs were developed for Western agriculture. For example, in Northern African countries, most crops, such as sorghum, wheat, and canola (oilseed rape), have wild weed relatives, thereby increasing the risk that genes from the herbicide-resistant crop varieties could be transferred to wild weed relatives.

The risk of herbicide-resistant genes from a transgenic crop variety being transferred to weed relatives has been demonstrated for canola (oilseed rape) and sugar beet.

Repeated use of herbicides in the same area creates problems of weed herbicide resistance. For instance, if glyphosate is used with HRCs crops on about 70 million ha, this might accelerate pressure on weeds to evolve herbicide-resistant biotypes. Sulfonylureas and imidazolinones in HRCs are particularly prone to rapid evolution of resistant weeds. Extensive adoption of HRCs will increase the hectareage and surface treated, thereby exacerbating the resistance problems and environmental pollution problems.

Bromoxynil has been targeted in herbicide resistant cotton by Calgene and Monsanto (Table 2). This herbicide has been used on winter cereals, cotton, corn, sugarbeets, and onions to control broad leaf weeds. Drift of bromoxynil has been observed to damage nearby grapes, cherries, alfalfa, and roses. In addition, leguminous plants can be sensitive to this herbicide, and potatoes can be damaged by it. Herbicide residues above the accepted standards have been detected in soil and groundwater, and as drift fallout. Rodents demonstrate some mutagenic responses to bromoxynil. Beneficial *Staphylinid* beetles show reduced survival and egg production, even at recommended dosages of bromoxynil. Crustaceans (*Daphnia magna*) have also been severely affected by this herbicide.

Toxicity of Herbicides and HRCs

Toxic effects of herbicides to humans and animals also have been reported. For example, the Basta surfactant (sodium polyoxyethylene alkyl ether sulfate) has been shown to have strong vasodilatory effects in humans and cardiostimulative effects in rats. Treated mice embryos exhibited specific morphological defects.

Most HRCs have been engineered for glyphosate resistance. Although adverse effects of herbicide-resistant soybeans have not been observed when fed to animals, such as cows, chickens, and catfish, genotoxic effects have been demonstrated on other non-target organisms. Earthworms have been shown to be severely injured by the glyphosate herbicide at

Table 2 Herbicide-resistant crops (HRCs) approved for field tests in the United States from 1987 to July 1995

| Crop | Herbicide | Research organization | | |
|-----------------------|-----------------------|--|----------------------------|--|
| Alfalfa | Glyphosate | Northrup King | | |
| Barley | Glufosinate/Bialaphos | USDA | | |
| Canola (oilseed rape) | Glufosinate/Bialaphos | University of Idaho Hoechst-Roussel/AgrEvo | | |
| | Glyphosate | InterMountain Canola Monsanto | | |
| Corn | Glufosinate/Bialaphos | Hoechst-Roussel/AgrEvo ICI UpJohn Cargill DeKalb Holdens Pioneer Hi-Bred Asgrow Great Lakes Hybrids Ciba-Geigy Genetic Enterprises | | |
| | | Glyphosate | Monsanto DeKalb | |
| | | Sulfonylurea | Pioneer Hi-Bred Du Pont | |
| | | Imidazolinone | American Cyanamid | |
| | | | Glyphosate | Monsanto Dairyland Seeds Northrup King |
| | | Cotton | Bromoxynil | Calgene Monsanto Rhone Poulenc |
| | | | Sulfonylurea | Du Pont Delta and Pine Land |
| | | | Imidazolinone | Phytogen |
| | | Peanuts | Glufosinate/Bialaphos | University of Florida |
| | | Potatoes | Bromoxynil | University of Idaho USDA |
| 2,4-D | USDA | | | |
| Glyphosate | Monsanto | | | |
| Imidazolinone | American Cyanamid | | | |
| Rice | Glufosinate/Bialaphos | | Louisiana State University | |
| | Glyphosate | | Monsanto | |
| Soybeans | Glyphosate | UpJohn Pioneer Hi-Bred Northrup King | | |
| | | Agri-Pro | | |
| | Glufosinate/Bialaphos | UpJohn Hoechst/AgrEvo | | |

(Continued)

Table 2 Herbicide-resistant crops (HRCs) approved for field tests in the United States from 1987 to July 1995 (*Continued*)

| Crop | Herbicide | Research organization |
|-------------|-----------------------|------------------------|
| Sugar Beets | Sulfonyleurea | Du Pont |
| | Glufosinate/Bialaphos | Hoechst-Roussel |
| | Glyphosate | American Crystal Sugar |
| Tobacco | Sulfonyleurea | American Cyanamid |
| Tomatoes | Glyphosate | Monsanto |
| | Glufosinate/Bialaphos | Canners Seed |
| Wheat | Glufosinate/Bialaphos | AgriEvo |

(From Refs.^[4-6].)

2.5–10.1/ha. For example, *Allolobophora caliginosa*, the most common earthworm in European, North American, and New Zealand fields, is killed by this herbicide. In addition, aquatic organisms, including fish, can be severely injured or killed when exposed to glyphosate. The beneficial nematode, *Steinernema feltiae*, a useful biological control organism, is reduced by 19–30% by the use of glyphosate.

There are also unknown health risks associated with the use of low doses of herbicides. Due to the common research focus on cancer risk, little research has been focused on neurological, immunological, developmental, and reproductive effects of herbicide exposures. Much of this problem is due to the fact that scientists may lack the methodologies and/or the diagnostic tests necessary to properly evaluate the risks caused by exposure to many toxic chemicals including herbicides.

While industry often stresses the desirable characteristics of their HRCs, environmental and agricultural groups, and other scientists, have indicated the risks. For example, research has shown that the application of glyphosate can increase the level of plant estrogens in the bean, *Vicia faba*. Feeding experiments have shown that cows fed transgenic glyphosate-resistant soybeans had a statistically significant difference in daily milk-fat production as compared to control groups. Some scientists are concerned that the increased milk-fat production by cows fed these transgenic soybeans may be a direct consequence of higher estrogen levels in these transgenic soybeans.

Economic Impacts of HRCs

Some analysts project that switching to bromoxynil for broadleaf weed control in cotton could result in savings of \$37 million each year. Furthermore, recent problems with use of glyphosate-resistant cotton in the Mississippi Delta region—crop losses resulting in up to \$500,000 of this year's cotton crop—suggest that this technology needs to be further developed before some farmers will reap economic benefits. In addition,

a recent study of herbicide-resistant corn suggests that the costs of weed control might be about two times more expensive than normal herbicide and cultivation weed control in corn.

While some scientists suggest that use of HRCs will cause a shift to fewer broad spectrum herbicides, most scientists conclude that the use of HRCs will actually increase herbicide use.

BT for Insect Control

More than 40 BT crystal protein genes have been sequenced, and 14 distinct genes have been identified and classified into six major groups based on amino acids and insecticidal activity. Many crop plants have been engineered with the BT α -endotoxin, including alfalfa, corn, cotton, potatoes, rice, tomatoes, and tobacco (Table 3). The amount of toxic protein expressed in the modified plant is 0.01%–0.02% of the total soluble proteins.

Some trials with corn demonstrate a high level of efficacy in controlling corn borers. Corn engineered with BT endotoxin has the potential to reduce corn borer damage by 5–15% over 28 million ha in the US, with a potential economic benefit of \$50 million annually. Some suggest that corn engineered with BT toxin will increase yields by 7% over similar varieties. However, it is too early to tell if all these benefits will be realized consistently. Potential negative environmental effects also exist because the pollen of engineered plants contains BT, which is toxic to bees, beneficial predators, and endangered butterflies like the Karka Blue and Monarch Butterflies.

Cotton was the first crop plant engineered with the BT α -endotoxin. Caterpillar pests, including the cotton bollworm and budworm, cost U.S. farmers about \$171 million/yr as measured in yield losses and insecticide costs. Benedict et al.^[3] predict that the widespread use of BT cotton could reduce insecticide use and thereby reduce costs by as much as 50% to 90%, saving farmers \$86 to \$186 million/yr.

Table 3 Transgenic insect resistant crops containing BT δ -endotoxins. Approved field tests in United States from 1987 to July 1995

| Crop | Research organization |
|-----------|---|
| Alfalfa | Mycogen |
| Apples | Dry Creek University of California |
| Corn | Asgrow Cargill Ciba-Geigy Dow Genetic Enterprises Holdens Hunt-Wesson Monsanto Mycogen NC+Hybrids Nortrup King Pioneer Hi-Bred Rogers NK Seed |
| Cotton | Calgene Delta and Pineland Jacob Hartz Monsanto Mycogen Northrup King |
| Cranberry | University of Wisconsin |
| Eggplant | Rutgers University |
| Poplar | University of Wisconsin |
| Potatoes | USDA Calgene Frito-Lay Michigan State University Monsanto Montana State University New Mexico State University University of Idaho |
| Rice | Louisiana State University |
| Spruce | University of Wisconsin |
| Tobacco | Auburn University Calgene Ciba-Geigy EPA Mycogen North Carolina State University Roham & Haas |
| Tomatoes | Campbell |

(Continued)

Table 3 Transgenic insect resistant crops containing BT δ -endotoxins. Approved field tests in United States from 1987 to July 1995 (Continued)

| Crop | Research organization |
|---------|---|
| | EPA |
| | Monsanto |
| | Ohio State University |
| | PetoSeeds |
| | Rogers NK Seeds |
| Walnuts | University of California, Davis USDA |

(From Refs.^{[4,7].})

The development of insect resistance to transgenic crop varieties is one highly possible risk associated with the use of BT δ -endotoxin in genetically engineered crop varieties. Resistance to BT has already been demonstrated in the cotton budworm and bollworm. If BT-engineered plants become resistant, a key insecticide that has been utilized successfully in IPM programs could be lost. Therefore, proper resistance management strategies with use of this new technology are imperative. Another potential risk is that the BT α -endotoxin could be harmful to non-target organisms. For example, it is not clear what potential effect the BT δ -endotoxin residues that are incorporated into soils will have against an array of non-target useful invertebrates living in the rural landscape. It has also been demonstrated that predators, such as the lacewing larvae (*Crysoperla carnea*) that feed on corn borers (*Ostrinia nubilalis*), grown on engineered BT corn have consistently higher mortality rates when compared to specimens fed with non-engineered corn borers. In addition, the treated larvae need three more days to reach adulthood than lacewings fed on prey from non-BT corn.

DISCUSSION

Both pesticides and biotechnology have definite advantages in reducing crop losses to pests. At present, pesticides are used more widely than biotechnology and thus are playing a greater role in protecting world food supplies. In terms of environmental and public health impacts, pesticides probably have a greater negative impact at present because of this more widespread use.

Genetically engineered crops for resistance to insect pests and plant pathogens could, in most cases, be environmentally beneficial because these more resistant crops could allow a reduction in the use of hazardous

insecticides and fungicides in crop production. In time, there may also be economic benefits to farmers who use genetically engineered crops; this will depend, however, on the prices charged by the biotechnology firms for these modified, transgenic crops.

There are, however, some environmental problems associated with the use of genetically engineered crops in agriculture. For example, adding BT to crops like corn for insect control can result in any of the following negative environmental consequences: 1) development of resistance to BT by pests species in corn and other crops; 2) health risks from exposure to the BT toxin to humans in their food and to livestock in feed; 3) the toxicity of the pollen from the BT-treated corn to honey bees, beneficial natural enemies, and endangered species of insects that feed on the modified corn plants or come into contact with the drifting pollen; engineered plant residues incorporated into soil can produce undesirable effects on soil micros and mesofauna.

A major environmental and economic concern associated with genetically engineered crops is the development of HRCs. Although in rare instances HRCs may result in a beneficial reduction of toxic herbicide use, it is more likely that the use of HRCs will increase herbicide use and environmental pollution. In addition, farmers will suffer because of the high costs of employing HRCs—in some instances, weed control with HRCs may increase weed control costs for the farmer threefold.

More than 40% of the research by biotechnology firms is focused on the development of HRCs. This is not surprising, because most of the biotechnology firms are also chemical companies who stand to profit if herbicide resistance in crops result in greater pesticide sales. Theoretically, the acceptance and use of engineered plants in sustainable and integrated agriculture should consistently reduce current use of pesticides, but this is not the current trend. In addition, most products and new technologies are designed for Western agriculture systems, not for poor or developing countries. For instance, if terminator genes enter into the seed market, there will be no possibility of traditional and small farmers using their plants to produce their seeds. Thus, genetic engineering could promote improvements for the environment; however, the current products—especially the herbicide-resistant plants and the BT-resistant crops—do have serious environmental impacts, similar to the consequences of pesticide use.

REFERENCES

1. James, C. Global Review of Commercialized Transgenic Crops: 1998. In *ISAAA Briefs*; Cornell University: Ithaca, New York, 1998; 8–1998.
2. Moff, A.S. Toting up the early harvest of transgenic plants. *Science* **1998**, *282*, 2176–2178.
3. Benedict, J.H.; Ring, D.R.; Sachs, E.S.; Altman, D.W.; DeSpain, R.R.; Stone, T.B.; Sims, J.R. Influence of Transgenic BT Cottons on Tobacco Budworm and Bollworm Behavior Survival, and Plant Injury. In *Proceedings Belt-wide Cotton Council*; Herber, J., Richter, D.A., Eds.; National Cotton Council: Memphis, Tennessee, 1992; 891–895.
4. Krinsky, S.; Wrubel, R.P. *Agricultural Biotechnology and the Environment*; University of Illinois Press: Urbana, Illinois, 1996.
5. McCullum, C.; Pimentel, D.; Paoletti, M.G. Genetic Engineering in Agriculture and the Environment: Risks and Benefits. In *Biotechnology and Safety Assessment*; Thomas, J.A., Ed.; Taylor & Francis: Washington, DC, 1998; 177–217.
6. Agribusiness. The Gene Exchange, Fall 1997; <http://www.ucsusa.org/Gene/F97.agribusiness.html> (accessed July 5, 2001).
7. Agribusiness. The Gene Exchange, Winter 1996; <http://www.ucsusa.org/Gene/W96.agribusiness.html> (accessed July 5, 2001).

BIBLIOGRAPHY

- Jepson, P.C.; Croft, B.C.; Pratt, G.E. Test systems to determine the ecological risks posed by toxin release from *Bacillus thuringiensis* genes in crop plants. *Mol. Ecol.* **1994**, *3*, 81–89.
- Lappè, M.; Bailey, B. *Against the Grain: The Genetic Transformation of Global Agriculture*; Earthscan Publication Ltd.: London, 1999.
- Paoletti, M.G.; Pimentel, D. The environmental and economic costs of herbicide resistance and host-plant resistance and host-plant resistance to plant pathogens and insects. *Technol. Forecast. Soc. Change* **1995**, *50*, 9–23.
- Paoletti, M.G.; Pimentel, D. Genetic engineering in agriculture and the environment. *BioScience* **1996**, *46* (9), 665–673.
- Pimentel, D.; Ali, M.S. An economic and environmental assessment of herbicide-resistant and insect/pest resistant crops. *Indian J. Appl. Econ.* **1998**, *7* (2), 241–252.
- Quaim, M. Transgenic Virus Resistant Potatoes in Mexico: Potential Socioeconomic Implications of North-South Biotechnology Transfer. In *ISAAA Briefs*; Cornell University: Ithaca, New York, 1998; 7–1998.
- Wrubel, R.P.; Gressel, J. Are herbicide mixtures useful for delaying the rapid evolution of resistance? A case study. *Weed Technol.* **1994**, *8*, 635–648.

Bird Control Chemicals

Eric B. Spurr

Department of Wildlife Ecology, Landcare Research New Zealand, Ltd.,
Lincoln, New Zealand

INTRODUCTION

Chemicals are used to control bird populations when they cause damage to crops, stock-food, buildings and other structures, and when they are hazards at places such as airports, public parks, golf courses, and rubbish dumps. They include lethal toxicants (avicides) and stressing agents, and non-lethal immobilizing agents, repellents, and reproductive inhibitors. Lethal methods of control attempt to reduce bird numbers, whereas non-lethal methods generally attempt to modify bird behavior without causing mortality, as a means of reducing damage.

TOXICANTS

The earliest toxicants were formulations containing arsenic, antimony, phosphorus, and various botanical extracts.^[1] Other toxicants used previously include chlorinated hydrocarbons such as endrin, metallic salts such as thallium, organometallic salts such as sodium monofluoroacetate (1080), alkaloids such as nicotine, and anticoagulants such as coumatetralyl and brodifacoum. Most are highly toxic to both birds and mammals. More than 2000 chemicals were evaluated as avicides between the 1940s and the 1980s, and some were found that were selectively toxic to birds. Some were even selectively toxic to certain species of birds. Since the 1980s, however, little effort has been put into finding new toxicants. Instead, most effort has been spent gathering toxicological and environmental data to ensure continued registration of existing products. Recently, international attention has focussed on the animal welfare aspects of toxicants.^[2]

Strychnine was once used widely as an oral toxicant for control of birds such as rock pigeons (*Columba livia*) and house sparrows (*Passer domesticus*), and is still used by certified operators in some countries today (Table 1). It is mainly applied in grain baits (e.g., Sanex Poison Corn in Canada). Strychnine is highly toxic to both birds and mammals, and poses a high risk of both primary and secondary poisoning to nontarget species. Time to death varies from 5 to 50 min. It causes extreme pain in poisoned animals and is considered inhumane.

Fenthion was previously used as an oral and dermal toxicant, but is currently used only as a dermal toxicant. Its use is restricted to certified operators. It is applied to wicks in artificial perches or other surfaces, for control of birds such as rock pigeons, house sparrows, and starlings (*Sturnus vulgaris*). It was available previously as Rid-A-Bird[®] in the United States, and currently as Control-A-Bird[®] and Avigrease[®] in Australia. Fenthion (Queletox[®]) is also aerially sprayed onto birds, especially red-billed quelea (*Quelea quelea*), in their nighttime roosts, to protect ripening grain crops in some African countries. It is highly toxic to birds and moderately toxic to mammals. Death occurs in 3 to 12 hr. The risk of nontarget bird mortality (from both primary and secondary poisoning) and environmental contamination is high, especially following aerial application. The symptoms of poisoning (e.g., convulsions) indicate that fenthion is likely to be inhumane.

4-Aminopyridine (Avitrol[®] in the U.S. and Canada, Avis Scare[®] and Scatterbird[®] in Australia) is often described as a frightening agent, but it is also an oral toxicant. Birds that ingest it die, but before dying they exhibit erratic behavior and alarm calling (often termed distress behavior) that supposedly frightens away other birds in the flock before they are able to ingest it. Time to death ranges from 15 min to 3 days. It is used to control birds such as rock pigeons, house sparrows, starlings, and in the U.S., red-winged blackbirds (*Agelaius phoeniceus*). It is available as a concentrate or as ready-to-use treated grain to certified operators. It is highly toxic to both birds and mammals, and may cause both primary and secondary poisoning of non-target species. Despite appearances to the contrary, it has been claimed that death from the compound is relatively painless. However, this needs to be verified because severe symptoms of intoxication may last for up to 3 days.

DRC-1339 (3-chloro-*p*-toluidine hydrochloride) (Starlicide[®]) is an oral toxicant used for the control of birds such as rock pigeons, starlings, and in the U.S., red-winged blackbirds. It is available as a concentrate or as a ready-to-use cereal-based bait to certified operators. Time to death varies from 3 to 50 hr, depending upon the amount of toxicant ingested. DRC-1339 is not suitable as a toxicant for all pest bird species because it is not highly toxic to all species. For example,

Table 1 Chemicals currently used for bird control in United States of America (U.S.A.), Canada, United Kingdom (U.K.), France, Israel, Australia, and New Zealand (N.Z.)

| Compound | Activity | Countries |
|-----------------------------|-----------------------------------|---|
| Strychnine | Oral toxicant | Canada, Australia |
| Fenthion | Oral and dermal toxicant | Some African countries, Australia |
| 4-Aminopyridine | Oral toxicant, frightening agent | U.S.A., Canada, Australia |
| DRC-1339 | Oral toxicant | U.S.A., N.Z. |
| Alpha-chloralose | Oral toxicant, immobilizing agent | U.S.A., France, U.K., Israel, Australia, N.Z. |
| Seconal (+alpha-chloralose) | Immobilizing agent | U.K. |
| Polybutene | Tactile repellent | U.S.A., Canada, U.K., Israel, Australia, N.Z. |
| Denatonium saccharide | Taste repellent | U.S.A., Canada |
| Aluminium ammonium sulfate | Taste repellent | U.K., Australia |
| Thiram | Taste repellent | France, Israel |
| Endosulfan | Taste repellent | France |
| Triacetate guazatine | Taste repellent | France |
| Methyl anthranilate | Irritant | U.S.A., Canada |
| Capsaicin | Irritant | U.S.A. |
| Naphthalene | Irritant | U.S.A. |
| Methiocarb | Secondary repellent | U.S.A., Canada, Israel, Australia, N.Z. |
| Ziram | Secondary repellent | U.K., France |
| Anthraquinone | Secondary repellent | U.S.A., France, N.Z. |
| Azacosterol | Reproductive inhibitor | Canada |
| Corn oil | Reproductive inhibitor | U.S.A. |
| Paraffin oil | Reproductive inhibitor | U.K. |

(Adapted from Refs.^[1-3] and Bibliography.)

it has only low toxicity to sparrows (Ploceidae) and finches (Fringillidae). It also has low toxicity to most mammals. This selective toxicity is unique. DRC-1339 is rapidly metabolized, so there is little risk of secondary poisoning. The death of birds from DRC-1339 has been described as painless, but symptoms such as difficult breathing indicate that this might not be so.

Alpha-chloralose is used in some countries (e.g., Australia and New Zealand) as an oral toxicant, but in other countries only as an immobilizing agent (see below). It is available to certified operators as a concentrate or as ready-to-use treated grain, for the control of birds such as rock pigeons and house sparrows. It is generally more toxic to birds than to mammals, and is relatively fast-acting. The first signs of narcosis may occur 10 min after ingestion, and immobilization may last for up to 27 hr, though it generally lasts less than 1 hr, after which birds may recover. However, death may result from hypothermia if sufficient active ingredient is ingested, and/or the weather is inclement. Alpha-chloralose is only slowly metabolized, and so may cause secondary poisoning of nontarget species. It is considered to be relatively humane on the basis of the generally short time to insensitivity.^[2]

LETHAL STRESSING AGENTS

PA-14 (Tergitol[®]) is a surfactant that was used as a lethal stressing agent in the U.S., but is no longer available for this purpose. It was sprayed onto birds, such as starlings and red-winged blackbirds, in their nighttime roosts, resulting in a break-down of the oil in the birds' feathers, destroying their natural waterproofing, and causing death from hypothermia.

IMMOBILIZING AGENTS

Immobilizing agents, administered in baits, are used to make birds easier to capture for removal from areas where they cause problems, or for killing humanely by other methods (e.g., by breaking their necks, or gassing them with carbon dioxide). Non-target birds that become immobilized can be revived and released. However, the effectiveness of immobilizing agents depends upon the amount ingested and environmental conditions. All known immobilizing agents are lethal to birds if they ingest a sufficient quantity. The most commonly used immobilizing agent worldwide is

alpha-chloralose, which is also used in some countries as a lethal toxicant (see above). In the U.S., it is available as an immobilizing agent only to approved operators, mainly to capture rock pigeons and waterfowl in nuisance situations. In the U.K., seconal is also used as an immobilizing agent, in combination with alpha-chloralose, to enhance its speed of action.

REPELLENTS

Chemical repellents can be primary or secondary in effect. Primary repellents are avoided reflexively because of an unpleasant sensation (e.g., touch, taste, smell, irritation). Tactile repellents include polybutene-based products (e.g., 4 The Birds[®], Hot Foot[®], and Tanglefoot[®] in the U.S., Bird-X, Buzz-Off[®], Shoo, Super Hunter, and Waco in Canada). They are applied to buildings and other structures, modifying the surface so that it becomes sticky or slippery and discouraging birds from landing or roosting. They are all available to the general public.

Taste repellents, which discourage birds from eating potential food sources to which they are applied, include denatonium saccharide (Ro-Pel[®] in the U.S. and Canada) and aluminium ammonium sulfate (Curb, Guardsman, and ReZist in the U.K., D-ter, Gaard, and Scat in Australia). Ro-Pel[®] also contains thymol, a fungicide that imparts a secondary repellent effect. Irritants include methyl anthranilate (ReJeX-iT[®] and Bird Shield[®] in the U.S., Avigon in Canada), capsaicin (Sevana), and naphthalene (Dr. T's), although there is no evidence that the latter two, by themselves, are effective.^[3] Methyl anthranilate may be applied to grassy areas such as parks and golf courses to deter feeding by birds such as Canada geese, and also to ripening fruit to deter birds such as house sparrows and starlings.

Secondary repellents cause post-ingestional illness, resulting in conditioned aversion to the treated food source. Examples include methiocarb (Mesuro[®]), ziram (AProtect), and anthraquinone (Flight ControlTM in the U.S., AvexTM in New Zealand). Methiocarb and ziram are moderately toxic to birds and mammals. In some countries, methiocarb may be applied to seeds and seedlings, but in the U.S. it may be used only in dummy egg baits to condition crows (*Corvus* spp.) not to prey on the eggs of endangered birds. Ziram and anthraquinone may be sprayed onto grass, field crops, ornamentals, conifers, and dormant fruit trees, but not onto products for immediate human consumption.

4-Aminopyridine is sometimes described as a frightening agent, and classified as a repellent, because it induces behavioral changes in birds. However, it is highly toxic to birds, and should be considered as a toxicant (see above).

REPRODUCTIVE INHIBITORS

Reproductive inhibitors have the potential to reduce bird populations by preventing or reducing the production of young. Azocosterol (Ornitrol[®]) is one of a number of chemicals that have been investigated for this purpose. It is applied to baits and fed to females daily for 10 to 15 days before egg-laying. It is no longer available in the U.S., but is still available for the control of rock pigeons in Canada. Corn oil (in the U.S.) and paraffin oil (in the U.K.) are two chemicals used to destroy the eggs of birds, such as gulls (*Larus* spp.) and Canada geese (*Branta canadensis*), after they have been laid. The oil may be sprayed onto the eggs in the nest, or the eggs may be temporarily removed, immersed in oil, and then returned to the nest. The oil occludes the pores in the shell, asphyxiating the developing embryo. The technique is considered humane.^[2]

FUTURE DEVELOPMENTS

No existing products are ideal for the control of pest birds. Toxicants are becoming increasingly publicly unacceptable worldwide from environmental and animal welfare perspectives. Currently, research is being done on the effectiveness of an oral toxicant/anaesthetic combination that reduces the time to unconsciousness, as a means of improving the animal welfare aspects of lethal bird control. Research is also being done on potential new repellents, including other derivatives of anthranilate, acetophenone, benzoate, cinnamamide, and d-pulegone. The use of nonlethal methods of bird control, especially repellents, may be a better option for the future than the use of toxicants.

REFERENCES

1. Schafer, E.W., Jr. Bird control chemicals—nature, modes of action, and toxicity. In *CRC Handbook of Pest Management in Agriculture*, 2nd Ed.; Pimentel, D., Ed.; CRC Press: Boca Raton, Florida, USA, 1991; Vol. 2, 599–610.
2. Ministry of Agriculture, Fisheries and Food. *Assessment of Humaneness of Vertebrate Control Agents*; Ministry of Agriculture, Fisheries and Food, Pesticides Safety Directorate: York, United Kingdom, 1997.
3. Clark, L. Review of bird repellents. In *Proceedings of the 18th Vertebrate Pest Conference*; Baker, R.O., Crabb, A.C., Eds.; University of California: Davis, California, USA, 1998; 330–337.

BIBLIOGRAPHY

Australian Ministry of Agriculture, Fisheries and Forestry, National Registration Authority for Agricultural and

- Veterinary Chemicals, Product database. <http://www.dpie.gov.au/nra/pubcris.html> (accessed July 2000).
- Hynstrom, S.E.; Timm, R.M.; Larson, G.E. *Prevention and Control of Wildlife Damage*; Nebraska Cooperative Extension Service, Great Plains Agricultural Council, and USDA-APHIS-Animal Damage Control: Lincoln, Nebraska, USA, 1994.
- Index Phytosanitaire ACTA 2000*, 36th Ed.; Association de Coordination Technique Agricole: Paris, France, 2000.
- Israel Ministry of Agriculture and Rural Development, Plant Protection and Inspection Services, Pesticides. Data Bank; <http://www.moag.gov.il/ppis/> (accessed September 2000).
- Ministry of Agriculture, Fisheries and Food, Pesticides Safety Directorate, and Health and Safety Executive. *Pesticides 2000*; The Stationery Office: London, United Kingdom, 2000.
- New Zealand Ministry of Agriculture and Forestry, Agricultural Compounds and Veterinary Medicines Group, ACVM database of currently registered pesticides. <http://www.maf.govt.nz/ACVM/reg-lists/index.htm/> (accessed July 2000).
- O'Connor, B., Ed.; *Novachem Manual 2000/2001: A New Zealand Guide to Agrichemicals for Plant Protection*; Novachem Services Ltd.: Palmerston North, New Zealand, 2000.
- Québec Ministère de l'Agriculture, des Pêcheries et de l'Alimentation, Les étiquettes de pesticides de l'ARLA. http://www.agr.gouv.qc.ca/dgpar/rap/html/rech_pdf_arla.html/ (accessed August 2000).
- United Kingdom Ministry of Agriculture, Fisheries and Food, Pesticides Safety Directorate, Product information. http://www.pesticides.gov.uk/raid_info/prod_inf.htm/ (accessed July 2000).
- United States Environmental Protection Agency, Pesticide product databases. <http://www.epa.gov/pesticides/products.htm/> (accessed July 2000).

Cabbage Diseases: Ecology and Control

Anthony P. Keinath

Coastal Research and Education Center, Clemson University,
Charleston, South Carolina, U.S.A.

Marc A. Cubeta

Center for Integrated Fungal Research, Plant Pathology, North Carolina State University,
Raleigh, North Carolina, U.S.A.

David B. Langston Jr.

Rural Development Center, University of Georgia, Tifton, Georgia, U.S.A.

INTRODUCTION

Cabbage (*Brassica oleracea* “Capitata Group”) has long been cultivated as an important vegetable crop and a source of vitamins, minerals, and fiber, particularly during cold seasons in temperate climates. More recently, cabbage and other cruciferous vegetables (members of the Brassicaceae) have been recognized as important sources of chemoprotective phytochemicals in the diet. Cabbage is a productive vegetable based on biomass per area of cultivation. However, this crop is affected by many diseases, particularly those caused by fungi and bacteria. This article focuses on six diseases of worldwide importance in cabbage production. These diseases also affect other cole crops, i.e., vegetables derived from *B. oleracea*, including broccoli, Brussels sprouts, cauliflower, collard, kale, and kohlrabi, and other genetically related cruciferous vegetables, such as turnip, rutabaga, Chinese cabbages, and mustards. Emphasis will be placed on stages in the life cycles of the pathogens that affect management. Control measures will be presented in an IPM context.

MAJOR DISEASES AND PATHOGEN ECOLOGY

Black Rot

Black rot is caused by the bacterium *Xanthomonas campestris* pathovar *campestris*. Because this bacterium can be seedborne, black rot is found in most areas of the world where cabbage and other crucifers are grown. The pathogen produces V-shaped chlorotic and necrotic lesions starting at the margins of leaves, but it also causes wilting of plants if it reaches the vascular system in the stem (systemic infection). Blackening of the leaf veins is a helpful diagnostic symptom. The pathogen survives in infested crop debris but can only live a few months in soil.

Clubroot

Clubroot is caused by the slime mold-like organism *Plasmodiophora brassicae*. This soilborne organism is an obligate parasite, completing its unique life cycle within the root cells of crucifers. Infected root cells enlarge and divide to produce the diagnostic swollen, club-like roots. The pathogen produces resting spores in the clubs that persist in soil for at least 10 years after the clubs decay. Isolates of *P. brassicae* differ in host range, and races have been found that are pathogenic on the few resistant cultivars of cabbage that have been bred.

Black Spot, Dark Leaf Spot

Two species of *Alternaria*, *A. brassicae* and *A. brassicicola*, infect cabbage and other crucifers. *A. brassicicola* has higher optimal temperatures for growth, sporulation, and spore germination (20–30°C.) than *A. brassicae* (18–24°C.). Both fungi can be seedborne and airborne, but do not survive apart from infested host debris in soil. Infested debris left on the soil surface can be a significant source of pathogen spores for up to 12 weeks after harvest.^[1] Seedborne inoculum can lower seed germination and vigor but usually is not damaging to seedlings.

Downy Mildew

Crucifer downy mildew is caused by the Oomycete *Peronospora parasitica*. This fungus-like organism produces airborne sporangia on leaf undersides and oospores inside infected tissues. The pathogen is believed to survive as dormant oospores in roots and stems. Cabbage is affected by downy mildew particularly during the seedling and heading growth stages. High relative humidity, dew, and fog are favorable for

infection. Separate host genes confer resistance in the cotyledon and adult plant stages of growth.^[2] Interactions observed between resistant plant varieties and isolates of the pathogen suggest that races of the pathogen exist.

Watery Soft Rot, Sclerotinia Stem Rot, White Mold

Sclerotinia sclerotiorum has a wide host range, but is especially damaging to cabbage, because it not only infects the head in the field but also can cause decay in storage. The common names for this disease show that infection occurs primarily on heads or stems of cabbage, particularly at maturity when wrapper leaves shade the soil, providing a cool, moist environment that favors the pathogen. This fungus produces airborne spores that infect plants, but soilborne survival structures (sclerotia) also can cause infection when they germinate near a plant.

Wirestem

Wirestem, a postemergence disease, is caused by the soilborne fungus *Rhizoctonia solani* anastomosis groups (AG) 4 and 2-1. In soils cropped repeatedly to crucifers, AG 2-1 predominates. At low pathogen levels, wirestem is more prevalent or more severe than preemergence damping-off. Seedlings may be killed by wirestem when lesions girdle stems. Older plants may be killed later as a result of seedling infections or be stunted and fail to produce a marketable-sized head. Root rot also occurs when infection is severe

but is absent when discrete stem lesions are the only symptoms.

CONTROL

General Control Principles

Exclusion

It is extremely important to prevent contamination of clubroot-free land by excluding the pathogen. Movement of transplants and equipment from clubroot-infested fields or farms should be avoided. Growers in clubroot-free areas should avoid purchasing field-grown transplants or equipment from infested areas.

Eradication

Outbreaks of black leg associated with seed have been reduced by testing seed for the pathogen *Phoma lingam*. Eradicate cruciferous weeds to eliminate sources of the pathogens causing black rot, downy mildew, and clubroot (Table 1). In addition, cruciferous ornamentals can be infected by the same species of *Alternaria*, *Peronospora*, *Plasmodiophora*, and *Xanthomonas* that infect cabbage.

Avoidance

Do not plant susceptible cabbage in pathogen-infested fields. Wirestem is less severe when cabbage is planted into cool soils than into warm soils. In addition, using a shallow planting depth for transplants avoids contact of the susceptible hypocotyl with *Rhizoctonia*-infested

Table 1 Management practices for common diseases of cabbage

| Disease | Plant resistant cultivars | Use healthy seed or transplants | Control weeds | Avoid wounding | Bury crop residue | Rotate with non-host | Apply protectant fungicide or bactericide |
|----------------------|---------------------------|---------------------------------|---------------|----------------|-------------------|----------------------|---|
| Black spot | + | + | + | – | + | + | + |
| Bacterial soft rot | – | – | – | + | – | – | – |
| Black leg | – | + | – | – | + | + | + |
| Black rot | + | + | + | + | + | + | –/+ |
| Clubroot | – | + ^a | + | – | – | – | +/- |
| Downy mildew | – | – | + | – | + | + | + |
| Yellows | + | + ^a | – | – | – | – | – |
| Sclerotinia stem rot | – | – | + | + | + | – | +/- |
| Damping-off | – | – | – | – | – | – | + |
| Wirestem | – | + ^a | – | – | + | – | + |

+, practice can be used to manage the disease.

–, practice is ineffective or inappropriate, based on the life cycle of the pathogen.

+/-, practice may be useful under certain conditions.

^aThe pathogen is not seedborne, but can be spread on infected, field-grown transplants.

soil. Avoid wounding plants to prevent black rot, bacterial soft rot, and watery soft rot.

Resistance

Host plant resistance is widely available in green (white) and red cabbage for yellows (caused by *Fusarium oxysporum* f. sp. *conglutinans*). Newer hybrid cultivars have partial resistance to black rot that restricts lesions to the wrapper leaves. A few cabbage cultivars (mostly red cabbage) have moderate resistance to *Alternaria*. Cabbage cultivars available in the U.S.A. are susceptible to *Sclerotinia*, downy mildew, wirestem, and clubroot.

Protection

Seed treatment is very effective to prevent damping-off caused by *Pythium* spp. and *R. solani*. Protectant fungicides are effective against foliar fungal pathogens and also are used against wirestem, clubroot, and black rot with varying degrees of success. Recently, the fungicide boscalid was registered in the U.S.A. to control *Sclerotinia* on cole crops.

Therapy

The only measure to control cabbage diseases post-infection is the application of systemic fungicides for downy mildew.

EXAMPLES OF INTEGRATED DISEASE MANAGEMENT

Controlling weeds, especially ragweed (*Ambrosia artemisiifolia*), can reduce incidence of watery soft rot. Ascospores of *Sclerotinia* infect ragweed flowers that then fall onto cabbage leaves and infect them, because flower parts provide nutrients for the pathogen.^[3] Control flea beetles (*Phyllotreta cruciferae*), which carry conidia of *A. brassicicola* on their bodies and in their frass and transmit conidia while feeding.^[4]

Private and public cabbage scouting programs have been developed and are useful for scouting production fields for diseases and insects. For example, the cabbage-scouting program in Suffolk County, New York, U.S.A., has operated for the past 20 years. In addition to insects, scouts record the presence, general severity, and field location of black rot, black spot, clubroot, downy mildew, viruses, watery soft rot, and yellows.

MANAGING SEEDBORNE PATHOGENS

Plant seed from seedlots that have tested negative for the presence of the pathogens that cause black rot and

black leg. Hot water seed treatment is useful to control seedborne black rot bacteria, provided the water temperature is monitored carefully so it remains at 50°C for 25 minutes. Minimize leaf wetness periods when producing transplants in glasshouses, because of the ease of spreading pathogens. Apply protectant fungicides to seed crops to prevent infection of seed by *Alternaria*.

MANAGING SOILBORNE PATHOGENS

Soil fumigants generally are not used against soilborne pathogens in cabbage production because of the high cost, although they may be used to disinfest seedbeds and suppress clubroot. Field-grown transplants may be sources of the wirestem and clubroot pathogens and spread them to non-infested fields. Because of this risk, transplants should be produced in soilless mixes in glasshouses when possible. Do not plant any cruciferous vegetables in fields before or after cropping to cabbage. Use monocots as rotation crops, because *R. solani* AG 4 has a wide host range among dicotyledonous crops. The resting spores of the clubroot organism cannot be eradicated by rotation. Instead, liming soil to raise the pH above 7.2 with calcium oxide or hydrated lime prevents infection of roots in many soils.

MANAGING FOLIAR PATHOGENS

Diseases caused by foliar pathogens, such as *Xanthomonas* and *Alternaria*, can be managed with crop rotation during the period when infested host debris is decaying in affected fields, because these foliar pathogens of cabbage do not survive longer than one or two years in soil, respectively. Disk and bury or compost unmarketable cabbage heads. Apply protectant fungicides as needed based on environmental conditions and host susceptibility. Because *Alternaria* spp. require relatively long periods of leaf wetness for infection (a minimum of five to nine hours), disease can be reduced by increasing row width and plant spacing to promote air circulation that dries leaves.

CONCLUSIONS

The diseases black spot, downy mildew, watery soft rot, and wirestem often can be managed successfully using a combination of cultural, biological, and chemical control measures. The cultural and biological methods listed in Table 1 also are amenable to organic production systems. Management of black rot and clubroot remains more challenging. In the future, resistance to downy mildew and improved resistance to black rot may be available in cabbage cultivars. It may

be possible to transfer downy mildew resistance from broccoli to cabbage using molecular genetics methods. Additional research is needed to clarify the identity of races of the downy mildew and clubroot organisms.

ACKNOWLEDGMENTS

Technical contribution No. 5100 of the Clemson University Experiment Station. We thank Richard Morrison, Sakata Seed Company, and J. Powell Smith, Clemson University, for reviewing this article.

ARTICLES OF FURTHER INTEREST

Dispersal of Plant Pathogens, p. 193.
Principles of Pest Management with Emphasis on Plant Pathogens, p. 666.

Vegetable Crop Pest Management (Plant Pathogens), p. 875.

REFERENCES

1. Humpherson-Jones, F.M. Survival of *Alternaria brassicae* and *A. brassicicola* on crop debris of oilseed rape and cabbage. *Ann. Appl. Biol.* **1989**, *115* (1), 45–50.
2. Coelho, P.S.; Monteiro, A.A. Expression of resistance to downy mildew at cotyledon and adult plant stages in *Brassica oleracea* L. *Euphytica* **2003**, *133* (3), 279–284.
3. Dillard, H.R.; Hunter, J.E. Association of common ragweed with Sclerotinia rot of cabbage. *Plant Dis.* **1986**, *70* (1), 26–28.
4. Dillard, H.R.; Cobb, A.C.; Lamboy, J.S. Transmission of *Alternaria brassicicola* to cabbage by flea beetles (*Phyllotreta cruciferae*). *Plant Dis.* **1998**, *82* (2), 153–157.

Cereals: Growing Systems and Pest Occurrence

Ján Gallo

Department of Plant Protection, Slovak Agricultural University,
Nitra, Slovak Republic

INTRODUCTION

From the economical, agronomical, and consumer points of view, cereals represent the most important group of crops throughout the entire structure of plant production. Occurrence and distribution of animal pests in cereals are not uniform and depend mainly on the climatic, agroecological, anthropogenic, and some other hardly definable factors. Introduction of various cereals' growing systems causes changes in occurrence of animal pests in these cultures. A lot of species which were found in our cereals in large amounts in the past (larvae of Elateridae—wireworms) have progressively become less significant, and on the contrary, species which were meaningless before (some *Diptera*) have been increasingly becoming significant animal pests recently. Relatively few authors are engaged in research on the total spectra of animal pests in cereals from the point of view of their occurrence and harmfulness. Influence of "new" growing systems on the occurrence of the pests and regulators has begun to be investigated only recently.

GROWING SYSTEMS OF CEREALS

Under our conditions, intensity of plant production as well as application of chemical control have decreased since the beginning of early 1990s as the consequence of restructuring and privatization of agriculture resulting (except other aspects) in the changes of species spectrum of animal pests.^[1,2] Very similar situation was registered in our country after the World War II during the transformation from extensive small-scale production to the large-scale production technologies connected with the specialization and concentration of the agricultural production. Introduction of simplified crop rotations and changes in structure of grown crops significantly affected changes in species spectrum of animal pests. Increased predisposition for higher occurrence of animal pests is also related to the growing high-yielding cultivars which were selected for reaching maximum yields under the application of high-growing intensity factors which, however, are missing in our conditions nowadays. In breeding and selection process, some cultivars have lost part of their

genus which were responsible for protective ability against animal pests. In long-run investigation of animal pests, it was found that decisive role is played by growing systems, whereas the effect of weather conditions is of secondary importance, although not vain. Only very extreme course of weather in respective year can contribute to the different gradation of the pests. Growing systems of cereals in the transition period would gradually result in sustainable plant production.^[3-5] Sustainable system is based on the potential of certain region and respects its limitations. This system neither exhausts local resources nor degrades living environment. It ensures reliable yielding of crops at competitive costs and optimal utilization of energy and materials.

Within this transition period, which has already lasted in this country over 10 years, we have been investigating an effect of various growing systems of cereals (spring barley, winter wheat) on occurrence and distribution of pests. The following growing systems there were analyzed: *biculture* (crop rotation: the 1st year: maize, the 2nd year: spring barley), *triculture* (crop rotation: the 1st year: maize, the 2nd year: winter wheat, the 3rd year: pea), *tetraculture* (crop rotation: the 1st year: maize, the 2nd year: spring barley, the 3rd year: pea, the 4th year: winter wheat), *ecological system* (crop rotation: the 1st year: spring barley, the 2nd year: alfalfa, the 3rd year: silage maize, the 4th year: winter wheat, the 5th year: pea, the 6th year: field bean and alfalfa as undercrop), which was based and investigated in accordance with the International Foundation for Organic Agriculture Movements (IFOAM) regulations, and *integrated system* (crop rotation: the 1st year: winter wheat, the 2nd year: silage maize, the 3rd year: winter wheat, the 4th year: pea, the 5th year: field bean and alfalfa as undercrop, the 6th year: spring barley). In addition, in each respective growing system, different soil cultivation (minimal and conventional) as well as differentiated nutrition (non-fertilized and balanced fertilization) were applied. The essential aim of this design was to maintain the low inputs of additional energy and respect ecological rules. Application of farmyard manure under the crops, which are usually manured by it, was substituted by plowing the postharvest residues down under these crops.

Monitoring of Animal Pests in Spring Barley

After winter wheat, spring barley is the second most important crop of our agriculture and still stands in the center of attention as an important raw material for malt and beer production, and, also, its suitability for animal feeding purposes is not negligible. Unlike winter wheat, spring barley by its habit creates higher precondition for being attacked by harmful organisms. Investigation of animal pest spectrum occurrence in spring barley which was included in biculture and tetraculture showed that the following species of animal pests are the dominant ones in both these growing systems: wheat trips (*Haplothrips tritici* Kurdjumov, 1912) and rye trips (*Limothrips denticornis* Haliday, 1836), cereal flea beetle (*Phyllotretavittula* L. Redtenbacher, 1849), frit fly (*Oscinella frit* Linnaeus, 1758), and straw fly (*Chlorops pumilionis* Bjerkander, 1778). In addition, under these conditions, the following three species of aphids have to be included as significant pests: *Sitobion avenae* Fabricius, 1775, *Metopolophium dirhodum* Walker, 1849, and *Rhopalosiphum padi* Linnaeus, 1758. Their significance is very high from the viewpoint of their harmfulness and, with it, related reduction in yields. At the beginning of the 1990s, *Agromyzidae*, mainly *Agromyza ambigua* Fallow, 1823 as well as *Agromyza megalopsis* Hering, 1933, and various bionid flies from the genera of *Bibio* and *Dilophus* still were being found. At the end of the decade, these species retrograde, and starting with the year 2000, they are continuously in the fallback, and various species of flea beetles from the genera *Phyllotreta* and *Chaetocnema* start to prevail. In recent years, a new barley aphid—Russian wheat aphid [*Diuraphis noxia* (Kurdjumov, 1913)]—has begun to appear in our conditions.

This experiment showed that occurrence of animal pests in biculture was higher (95 pieces per 5 m²) than in tetraculture (78 pieces per 5 m²). This fact would be seen particularly in relation to structure of the crops. In the course of investigation, some influence of nutrition on animal pest occurrence was recognized. Occurrence of pests was lower (73–53 pieces per 5 m²) on unfertilized treatments in comparison with fertilized ones (83–62 pieces per 5 m²) in both systems (biculture and tetraculture). Explanation is attributed to the fact that fertilized plants have finer tissues, and, for that, they are more frequently visited by animal pests. In both systems (biculture and tetraculture), some effect of soil tillage was also detected. In biculture, treatment, which was cultivated in conventional way, showed lower occurrence of pests (63 pieces per 5 m²) compared to treatment with minimal soil tillage (82 pieces per 5 m²). On the contrary, in tetraculture, higher pest occurrence was under conventionally cultivated treatment (67 pieces per 5 m²) than under

minimally cultivated one (50 pieces per 5 m²). These results revealed distinct effect of soil cultivation on pest occurrence.^[6]

Spring barley grown in ecological and integrated cropping system confirmed the fact we theoretically predicted more than 10 years ago. Long-term investigation approved that at the beginning of 1990s, counts of animal pests were higher in spring barley grown in integrated system (68 pieces per 5 m²) than in ecological one (48 pieces per 5 m²). After more than 10 years, the situation has changed completely. In recent years, spring barley has been more intensively invaded by pests in ecological system (105 pieces per 5 m²) compared with integrated system (90 pieces per 5 m²). This phenomenon is closely related to the diversity of the animal pests and their broader species spectrum in ecological growing system. In both ecological and integrated systems, the same species composition of dominant animal pests as in biculture and tetraculture was found out.

Monitoring of Animal Pests in Winter Wheat

Winter wheat is our most important “bread” crop and represents the essential source of human nutrition. It appeared as a model crop in the process of application and utilization of scientific agrotechnical methods on one side and as a typical crop from the viewpoint of intensification of agricultural production on the other side. In this experiment, winter wheat was growing in triculture and tetraculture. In both these growing systems, the dominant animal pests were represented by the following species: wheat trips (*H. tritici*) and rye trips (*L. denticornis*), cereal flea beetle (*P. vittula*), brassy flea beetle (*Chaetocnema concinna* Marshall, 1802), lema black cereal beetle (*Oulema melanopus* Linnaeus, 1758), lema blue cereal beetle (*Oulema gallaeciana* Heyden, 1870), frit fly (*O. frit*), and straw fly (*C. pumilionis*). In addition, the following three species of aphids have to be included as significant pests of winter wheat: *S. avenae*, *M. dirhodum*, and *R. padi*. Significance of these species is very high because they reduce yields of winter wheat grain when they are present in the wheat cover. Thus species composition of dominating animal pests is only slightly different from that determined in spring barley. *Agromyzidae*, mainly *A. megalopsis*, various bionid flies from the genera of *Bibio* and *Dilophus*, *Cnephasia pumicana* Zeller, and sawflies (*Dolerus gonager* Fabricius, 1781 and *Dolerus haematodes* Schrank, 1781) were still being found in winter wheat (likely in spring barley) at the beginning of the 1990s. At the end of the decade, these species retreat and a new pest—aphid (*D. noxia*) starts to appear in the winter wheat covers.

The results of this experiment showed that winter wheat grown in triculture was more infested by animal pests (85 pieces per 5 m²) than that grown in tetraculture (68 pieces per 5 m²). In both these growing systems, fertilization markedly influenced pest occurrence. In the treatments without fertilizing, the occurrence of animal pests was lower (74 and 64 pieces per 5 m²) than in fertilized variants (84 and 78 pieces per 5 m²) in triculture and tetraculture growing systems, respectively.

Winter wheat was also differentially attacked by pests when grown in ecological or integrated growing system. More animal pests were counted in ecological system (87 pieces per 5 m²) in comparison with integrated system (71 pieces per 5 m²).

During the investigating period, spring barley was more intensively infested by pests (115 pieces per 5 m²) than winter wheat (69 pieces per 5 m²) on the average.

On the average of experimental period (more than 10 years), the maximum infestation of animal pests in spring barley was within the period of the 1st 10 days of May (259 pieces per 5 m²) and then still within the period of the 1st 10 days of June (242 pieces per 5 m²). In winter wheat, the maximum infestation was observed within the period of the 2nd 10 days of May (166 pieces per 5 m²) and then within the period of the 1st 10 days of June (159 pieces per 5 m²).

CONCLUSION

Investigated experimental growing systems of cereals (biculture, triculture, tetraculture, ecological system, and integrated system) showed differences in occurrence of animal pests in both spring barley and winter wheat. Species composition of dominating animal pests was very similar in these crops and included the following species: *H. tritici*, *L. denticornis*, *P. vittula*, *O. frit*, *C. pumilionis*, *S. avenae*, *M. dirhodum*, *R. padi*,

O. melanopus, *O. gallaeciana*, and *D. noxia*. Both cereals were more infected by animal pests when grown in the systems with simpler crop rotation (biculture and triculture) or with absence of chemical control (ecological system). Occurrence of animal pests in spring barley grown within biculture was higher than that in tetraculture. Winter wheat grown in triculture was more infected by animal pests than in tetraculture. Under ecological growing system, both spring barley and winter wheat were more infected by animal pests in comparison with integrated system. In general, during the investigating period, spring barley was more attacked by pests than winter wheat on the average.

REFERENCES

1. Markovec, A.F.; Gorbunová, N.N. Nasjakomyja, jakija poškozvajuc chlebnije zlaki u BSSR (Insects damaging wheat in Belarus). Vesni AN BSSR **1951**, *2*, 80–87.
2. Šedivý, J.; Kodys, F. Usměrněná chemická ochrana proti škůdcům obilnín (Regulated chemical control of grain crop pests). Metod. Závád. Výsled. Výzk. Praxe **1985**, *č. 1*, 43 s.
3. Gallo, J.; Pekár, S. Winter wheat pests and their natural enemies under organic farming system in Slovakia: effect of ploughing and previous crop. Anz. Schädlingsskd./J. Pestic. Sci. **1999**, *72*, 31–36.
4. Gallo, J.; Pekár, S. Effect of ploughing and previous crop on winter wheat pests and their natural enemies under integrated farming system in Slovakia. Anz. Schädlingsskd./J. Pestic. Sci. **2001**, *74*, 60–65.
5. Pfiffner, L. Effects of different farming systems on the presence of epigeal arthropods, in particular of carabids (*Col.*, *Carabidae*), in winter wheat plots. Mitt. Schweiz. Entomol. Ges. **1990**, *63*, 63–76.
6. Samsonová, I.V. Effect of shallow soil cultivation on numbers of spring wheat pests. Naučno-teh. Bùll. RASKhl. **1991**, *2*, 34–37.

Chemigation

Patricia S. Larraín

Carlos E. Quiroz

Instituto de Investigaciones Agropecuarias, Centro Regional de Investigación INIA-Intihuasi,
La Serena, Chile

INTRODUCTION

Increasingly more stringent constraints on food production are forcing farmers to search out new ways to produce food. This is especially so for the practice of pest management because the farmers are being compelled to reduce the air, soil, and water contaminants, preserve, augment, and enhance the biological control efforts, reduce the pesticide residues to safe levels, eliminate wastes, and protect the health of both the laborers and consumers.

Insecticides are often applied as foliar sprays or soil treatments. Foliar sprays can drift in air currents, resulting in the unnecessary exposure of the natural enemies and non-target organisms, while soil treatments can result in surface and subsurface runoff, translocating pesticides and thereby contaminating both the soil and groundwater.

Environmental stewardship dictates that the alternative methods be used. One such practice, currently becoming more widespread, is chemigation, the application of chemicals through irrigation systems.

WHY CHEMIGATION?

Chemigation is the process whereby agricultural chemicals, pesticides, and fertilizers are applied to soils and crops using, as the vehicle, water within an irrigation system [Note: The reviewer points out that “chemigation” also includes fertilizer that is run through an open ditch and is non-pressurized.][¹] Thus “chemigation” denotes any device or the combination of devices that utilize a hose, pipe, or other conduit to deliver a mixture of water and chemical directly to an agricultural system. Chemigation can comprise of a system consisting of an irrigation pumping station, a chemical injection pump, a reservoir for the chemical, a calibration device, a backflow prevention device, and a related safety equipment[²] (Figs. 1 and 2). Chemicals injected into the irrigation lines must be soluble in water.

Chemigation initially involved only the plant nutrients that generally require incorporation into the soil

to be effective. However, the concept of chemigation is expanding as a result of the advances in irrigation system design, improved chemical injection equipment, new agricultural pesticides formulated for use in chemigation systems, and refined pest management techniques. Nowadays, chemigation includes the applications of fertilizers, herbicides, insecticides, fungicides, nematocides, plant growth regulators, and biorationals.

Chemigation requires a well-designed and maintained equipment and people who will operate it that are well trained to ensure that the chemicals are applied at the right time and in the correct amounts. Although pressurized irrigation systems are more complicated to operate than non-pressurized irrigation systems, they facilitate chemigation. Chemigation requires a relatively high level of management. These and other advantages and disadvantages of chemigation are described in Table 1.[³]

CHEMIGATION AS AN INTEGRATED PEST MANAGEMENT (IPM) TOOL

Insecticides were first applied by chemigation in the mid-1970s. The method was, at that time, termed insectigation. The first trials, applied through pivot-attached sprayers, were conducted with methomyl to control the fall armyworm, *Spodoptera frugiperda*. This application method was more successful when the compounds were formulated in an oil base.[⁴] Emulsifiable concentrate (EC) and oil (EC plus non-emulsifiable oil) formulations of chlorpyrifos chemigated at two different volumes were compared with the soil-incorporated chlorpyrifos at the time when sweet potatoes were planted;^[5] the differences in total wireworm feeding damage were not statistically significant: all treatments resulted in significant reductions in feeding damage compared with the nontreated check. A greater reduction in the wireworm damage occurred with chemigation at higher water rates, possibly as a result of a better penetration of the insecticide into the upper soil layers. Oil-containing formulations applied aerially tend to reduce wash-off from leaf surfaces, while those applied directly to the soil need not be formulated with oils.[⁵]

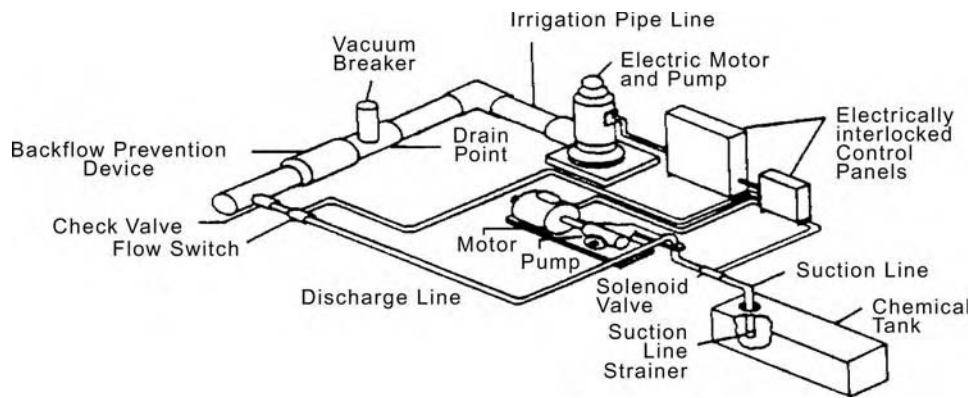


Fig. 1 Safety devices for the injection of chemicals into the irrigation systems having electric power. (Source: From Ref.^[2].)

Insecticide application through the center-pivot irrigation systems was found to be as efficient as the application with a high-clearance sprayer in controlling the corn earworm and fall armyworm larvae in the fresh-market sweet corn.^[6] Properly sized and located hollow cone and rotating irrigation sprinklers along the length of the center pivot resulted in a uniform pesticide application with <3% corn ear damage, compared with >57% damage in the untreated plots. Chemigation through the center pivots can control the insect pests in sweet corn and cotton as effectively as the conventional application methods.

To date, a distinction is made between the application of pesticides in irrigation water (chemigation) and through pivot-attached sprayer systems (PASS).^[7] Within chemigation, center-pivot irrigation systems or sprinkler irrigation are commonly used for the control of soil insect pests or chewing insect pests. However, chemigation is not only confined to irrigation systems

that apply water above the crop canopy. Chemigation methods have adapted to the major changes that have occurred in the irrigation industry during the last 2 decades. Although drip irrigation started about 30 years ago, recent advances in traditional drip irrigation, microirrigation, and subsurface drip irrigation have dramatically improved the efficiency of the systems and have allowed the system to expand immensely in area coverage. In many areas of the world, especially in the arid regions, the predominant mode of irrigation has switched from surface to drip.

Not surprisingly, chemigation is becoming widely used in drip irrigation systems, especially for sucking pests, because of the rapid uptake by the plants of chemicals that possess high biological activity to this group of pests.

The effects of the imidacloprid formulation and soil placement were tested on the sweet potato whitefly.^[8] Soil surface applications at 4-cm subseed furrow,

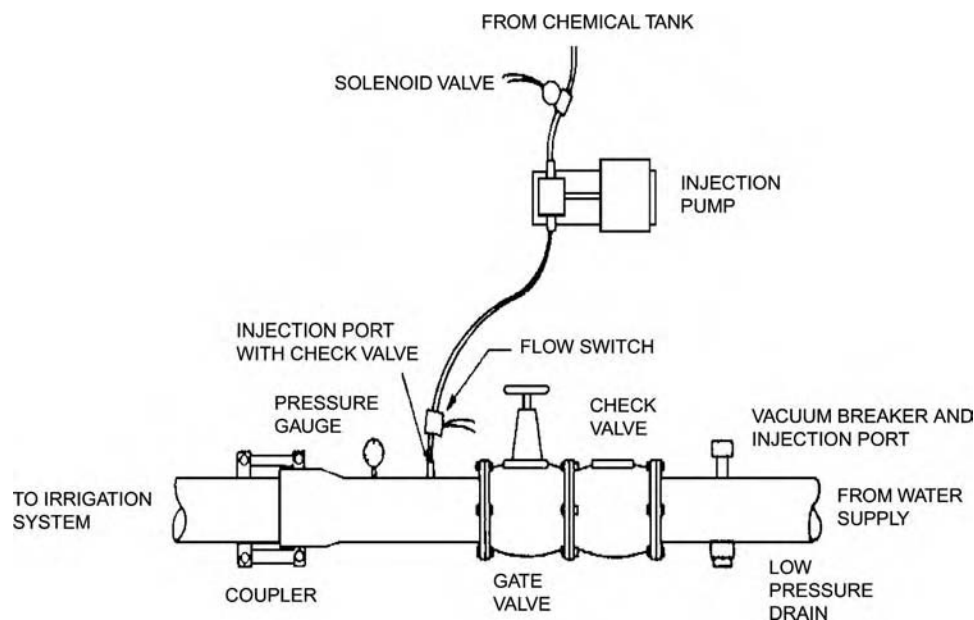


Fig. 2 Backflow prevention device using the check valve with vacuum relief and low-pressure drain. (Source: From Ref.^[2].)

Table 1 Advantages and disadvantages of chemigation as a method of pest management

| Advantages | Disadvantages |
|--|--|
| Properly designed and calibrated irrigation systems ensure a more uniform application | Chemigation requires highly efficient equipment and may require additional equipment |
| Chemical aids effectiveness of agricultural chemicals that require moisture for the activation or precise depth of incorporation | Chemigation requires considerable management input and personnel training |
| It reduces soil compaction by limiting the need for tractors in the field | It faces some environmental concerns if the chemicals are not correctly used, such as contamination of groundwater |
| Chemigation reduces operator exposure to chemicals | Not all pesticides are labeled/formulated for chemigation which may limit the management choices |
| It can reduce spray drift as well as water and soil contamination if the application is regulated with precision | |
| Chemigation can reduce the grower's chemical application costs and energy consumption for application by 90% | |
| In some cases, chemigation can eliminate the need for soil incorporation | |
| Drip chemigation conserves biological control species as well as any other nontarget species living above the soil | |
| Chemigation reduces waste and avoids above tolerance maximum residue limit (MRL) residues | |

followed by irrigation, provided the most consistent control of nymphs in small plots and in on-farm lettuce plots. From these sorts of experiments emerged the idea of applying properly the formulated insecticides directly via drip irrigation. Testing imidacloprid application rates and methods to control the grape mealybug, *Pseudococcus viburni* (Signoret), on table grapes, one application in the spring through drip irrigation at rates of >0.75 grams of active ingredient (g.a.i.) per plant was found to provide effective control for the entire season and for up to two seasons if the population pressures remained low.^[9]

Chemigation can also be applied through subirrigation, a system that is increasingly being used to water and fertilize greenhouse crops. Chemigation also provides a means of delivering systemic pesticides. Experiments testing the interactive effects of the modes of application and irrigation of imidacloprid to control whiteflies on poinsettias demonstrated that subirrigation delivered better protection between 8 and 10 weeks post-application than did drip irrigation. Similarly, whiteflies reproduced less, resulting in fewer immatures when the chemical was applied to the bottom of the subirrigated pots than when applied as a drench to drip-irrigated plants.^[10]

Soil distribution, plant uptake, and efficacy of imidacloprid applied through drip chemigation to control aphids in commercial hop yards were also studied.^[11] In this study, the aphid control was excellent, with movement of imidacloprid up to 90 cm within the irrigation system and residues at harvest below the U.S. tolerance of 6 ppm.

Drip chemigation is an environmentally suitable and more effective alternative to foliar-applied insecticides for the control of sucking pests. Part of the benefit of using chemigation is the elimination of the spray drift exposure, excessive environmental contamination, and the direct effects of insecticides on beneficial and non-target organisms.

CONSIDERATIONS IN THE USE OF CHEMIGATION

An important aspect to be considered for environmental safety is the uniformity of application within the irrigation system. Although no irrigation system is able to distribute water evenly over all locations, it is important that variations be kept within reasonable limits for proper water and chemical distribution. Chemigation is not the same thing as irrigation, so avoid overapplication, which is expensive and may well prevent the development of pest resistance. Chemigation is an excellent means for delivering a pesticide, but it must always be carefully managed.

The conditions of the soil are also known to be important; most pesticides should not be applied to wet or saturated soil. This can result in surface runoff, increasing the likelihood of surface water contamination. In this sense, the least amount of water possible should be among the guidelines followed to assure the proper pesticide application.

Drift is also another important consideration, especially for center pivot, linear move, PASS, and

similar delivery systems. Proper nozzle sizes and water pressures are needed to ensure large water droplets, which are less prone to wind drift.^[7]

Chemigation can be a safe and effective method for pest management provided that the system is properly designed and well operated and that the safety precautions are followed. Among the safety measures, backflow prevention is one of the most important. Legislated controls have been in place in many areas since the 1980s. In the United States, several states are requiring the use of backflow prevention or "chemigation valves" that are designed to stop the water and chemical mixture from draining or siphoning back into the irrigation water source.^[12] Effective April 1988, the U.S. Environmental Protection Agency regulations require labeling for each pesticide approved for application through irrigation systems. Labels also have information on procedures and restrictions.^[13] Local regulation should be followed in selecting and using chemigation equipments and procedures.

CONCLUSION

Chemigation, or application of agricultural chemicals to soils and crops using water within an irrigation system, is a method increasingly used in world crops.

The concept of chemigation is expanding as a result of the advances in irrigation system design, improved chemical injection equipment, new agricultural systems, and refined pest management techniques. Thus, the efficiency in plant uptake of chemicals to the soil has been improved, as well as the efficiency of distribution when applied directly to plant canopy, in both cases using the properly designed and celebrated irrigation system.

Chemigation is an excellent means for delivering a pesticide and can perfectly be highly recommendable in IPM programs, but it must always be carefully managed. Considerations in the use of chemigation are described in this article.

FUTURE PROSPECTS

Many aspects of chemigation have to be investigated to ensure the reliability of the method. For example, the pesticide residues in soils have an increased potential for leaching and runoff under yet undefined conditions. How leaching and runoff are influenced by soil texture and soil humidity must be addressed. The speed and quantity of pesticide uptake and movement

by different plant species are another areas ripe for investigation. Also needed is the information on the efficiency of the various chemigation methods in relation to the buildup of residues at harvest.

Results thus far strongly suggest that chemigation is an excellent and reliable method for pest control, but precautions must be taken by farmers. Researchers must delve deeper into the subject to ensure that chemigation is a truly safe, reliable, and efficient IPM tool.

REFERENCES

1. <http://www.ag.state.co.us/DPI/publications/chem.html> (accessed January 2002).
2. American Society of Agricultural Engineers (ASAE). *Safety Device for Chemigation. EP409.1*; ASAE: St. Joseph, MI, 1998; 880–882.
3. http://www.irrigation.org/about_common_chemigation.htm (accessed March 2002).
4. Johnson, A.W.; Young, J.R.; Threadgill, C.C.; Sumner, D.R. Chemigation for crop production management. *Plant Dis.* **1986**, *70* (11), 998–1004.
5. Chalfant, R.B.; Bondari, K.; Sumner, H.R.; Hall, M.R. Reduction of wireworm (Coleoptera: Elateridae) damage in sweet potato with insecticides applied by chemigation. *J. Econ. Entomol.* **1992**, *86* (1), 123–130.
6. Chandler, L.D.; Sumner, H.R.; Mullinix, B.G. Assessment of irrigation-related insecticide application methods for control of lepidopteran insect pests of sweet corn. *J. Econ. Entomol.* **1994**, *87* (1), 212–219.
7. <http://www.nal.usda.gov/ttic/tektran/data> (accessed January 2002).
8. Palumbo, J.C.; Kerus, D.L.; Engle, C.E.; Sanchez, C.A.; Wilcox, M. Imidacloprid formulation and soil placement effects on colonization by sweetpotato whitefly (Homoptera: Aleyrodidae): head size and incidence of chlorosis in lettuce. *J. Econ. Entomol.* **1996**, *89* (3), 735–742.
9. Larraín, P. Efecto de la quimigación y el pintado con imidacloprid sobre la población de *Pseudococcus viburni* (Signoret) (Homoptera: Pseudococcidae) en vides de mesa. *Agric. Tec. (Chile)* **1999**, *59* (1), 1–12.
10. Van Iersal, M.W.; Oetting, R.D.; Hall, D.B.; Kang, J. Application technique and irrigation method affect imidacloprid control of silverleaf whiteflies (Homoptera: Aleyrodidae) on poinsettias. *J. Econ. Entomol.* **2001**, *94* (3), 666–672.
11. Felsot, A.; Evans, R.; Tallman, L. Soils distribution and plant uptake of imidacloprid under drip and furrow irrigation. *Proceedings of the 4th Decennial Symposium, Phoenix, AZ, Nov 14–16, 2000*; 416–427.
12. <http://cati.csufresno.edu/cit/rese> (accessed March 2002).
13. <http://es.epa.gov/techinfo/facts/nc/nc-fs1.html> (accessed January 2002).

Chemistry of Pesticides

J. K. Dubey

Department of Entomology, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India

S. K. Patyal

Department of Entomology and Apiculture, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India

INTRODUCTION

The term pesticides is an all-inclusive word meaning killer of the pests (the ending “cide” comes from the Latin “cida”, meaning killer). Pesticides are legally classed as economic poisons and are defined as any substance used for controlling, preventing, destroying, or mitigating pest.^[1] In this article, the chemistry of current major pesticides is discussed.

WHY THE EVALUATION OF PESTICIDES?

Evaluation of pesticides includes the efforts to determine the tolerance levels of pesticide in man and is concerned with establishing logical basis for selective toxicity, in order to kill the pests without harming human beings and domestic animals. The suitability of pesticides is judged on the basis of bioactivity, safety, and toxicological parameters, viz., LD₅₀, LC₅₀, ED₅₀, MRL, ADI, etc. Higher concentration of all the pesticides are invariably toxic to most biological systems, but such concentration of pesticides cannot be used. Therefore, it is necessary to find a suitable dose that could bring about the desired effect. The current world consumption of pesticides is as follows: 43% herbicides, 32% insecticides, 19% fungicides, with the remaining 6% divided between growth regulators and miscellaneous agrochemicals. Organophosphates (OPs), carbamates and pyrethroids have almost replaced the more persistent organochlorines (OCs).^[2] Pyrethroid insecticides are safer and effective than OPs and OCs, requiring less active ingredient; as a result, the real poundage declined between 1976 and 1982, but treated acreage stabilized, and, alternatively, the use of herbicides has increased.^[2] The size of the global market in 1995 was estimated at \$35 billion, in which generics market size was around \$17.5 billion or 53%; and by the end of 2000, this is expected to grow to \$27 billion or 70% of the total, making a 54% expansion in 5 years growing at an annual rate of 9%.^[3] The emphasis has clearly been shifting to the chemistry of pesticides in order to understand biological and biochemical

pathways, and on achieving economical viable and environmentally safe methods of mitigating pests and diseases that ravage plant and animal life to the detriment of human welfare.

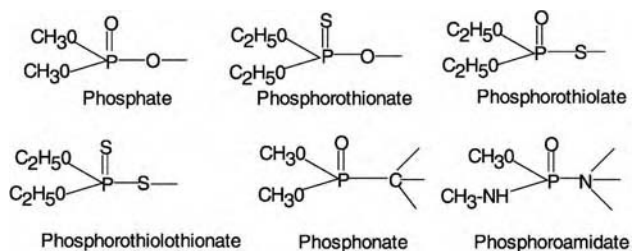
We evaluate the pesticides in order to calculate the risk involved and to determine whether the gain in their usage is significant enough to justify their use. If we were to say that we must be sure that absolutely no risk is involved in using pesticides, every pesticide would have to be withdrawn and no new pesticides would ever be introduced. Evaluation and testing of pesticides include laboratory studies and small, medium, and large-scale field studies to determine the safety, efficacy, ease of application, acceptability, and cost effectiveness. Continued evaluation of pesticides is a key part of monitoring resistance in the field population to order to provide data on which sound management decision can be made. It is therefore desirable that only bioactive, biodegradable, and eco-friendly pesticides should be used for pest control operation. The intermediate step between laboratory screening and field trials is crucial to studying a range of factors, such as dosage rate, formulation changes, and application parameters in order to limit the number of treatments that have to be taken through the later stages of evaluation under practical conditions.

USES OF PESTICIDES

1. Pesticides are not only practical and effective in controlling almost all pests species attacking plants, animals, man, stored raw, and processed products, etc.
2. Pesticides give positive and rapid pest control. Explosive increases in pest populations are reduced to below the economic threshold levels within hours of applications.
3. Pesticides can be used to meet emergency situations (pest outbreak) and prevent further losses.
4. Control is economical; pest complexes can be controlled by using compatible formulation of different pesticides like insecticides and fungicides or herbicides.

CHEMISTRY OF PESTICIDES

In order to understand the biochemical reactions, it is necessary to have some basic knowledge of organophosphorous insecticides, which are esters of phosphoric acid. These insecticides are generally acutely toxic to man and vertebrate animals and are non-persistent. Most of them have short residual activity, which is desirable in keeping down on food crops, but is often a problem when longer persistence is required. Martin^[4] gave the following schemes (Scheme 1):



Scheme 1

Phosphorus esters have the distinctive combinations of oxygen, carbon, sulfur and nitrogen attached to the phosphorus atom. The names of the phosphor molecules above are a part of much more complicated chemical names for the compounds that contain these building blocks (moieties). There are three chemical classes of organophosphorus insecticides, namely:

- Aliphatic derivatives having a carbon chain structure.
- Phenyl derivatives containing benzene ring with one hydrogen atom in the ring being replaced by the phosphorus moiety.
- Heterocyclic derivatives also containing a ring structure and phosphorus group. In a heterocyclic carbon ring, however, oxygen, nitrogen, or sulfur displaces one or more carbon atoms and the ring may consist of three or five or six rings.

Aliphatic Organophosphates

The oldest and best known insecticide of this class is malathion. This insecticide has low mammalian toxicity and is effective against many insect pests, and it has been in large-scale use for almost 40 years in agriculture and public health, and for the control of storage and household pests. Several aliphatic OPs, such as monocrotophos, demeton, and dimethoate, have a systemic action and are effective against sucking insect pests. Some insecticides, although highly toxic, have such a short residual activity that they are preferred for use on vegetable crops. Examples listed hereunder

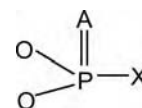
include acephate, demeton, demeton-methyl, dichlorvos, dimethoate, malathion, methamidophos, mevinphos, monocrotophos, oxy-demeton methyl, phorate phosphomidon, and trichlorfon.

Phenyl Organophosphates

These compounds are generally more stable than the aliphatic ones and are more persistent. Methyl parathion is slightly less toxic than other older members of this class, such as fenitrothion and tetrachlorvinphos, which are widely used in agriculture, horticulture, and public health programs. Included in this group are: bromophos, fenthion fensulfothion, fenitrothion, parathion methyl, phosalone, and triazophos, etc.

Heterocyclic Organophosphates

All the organophosphorus insecticides have the following general formula (Scheme 2):



Scheme 2

Where A is oxygen or sulfur, and RO and R'O are alkoxy groups which are usually either O,O diethyl or O,O dimethyl. In most of the commercial insecticides they are identical. X is the acidic group which is specially subjected to chemical attack because of a positive charge developing on the phosphorus atom. For this reason, it is often known as "the leaving group." The electrophilic nature of phosphorus depends upon the nature of leaving group X.

Although the nature of group X is principally responsible for the activity of organophosphorus insecticides, if this group is electrophilic, it tends to draw electrons away from P thereby creating a positive site in the vicinity of the phosphorus atom. The creation of positive nature on this site facilitates the hydrolysis of the compound. Although the nature of group X is principally responsible for the activity of the organophosphorus insecticides, the other radicals cannot be ignored. For example, an analog of parathion is biologically much less active than that of parathion despite having the same acidic or leaving group.

The biological activity of organophosphorus insecticides, therefore, does not depend only on the chemical properties of the compound but also on the steric factors. It is the reason why the biochemical interactions of the molecules are more pronounced, wherein the

molecule blocks the activity of the center of the esterase by fitting (irreversibly) into them.

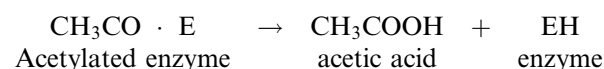
Parathion and malathion are poor inhibitors of cholinesterase *in vitro*, but they are very efficient with cholinesterase *in vivo*. The reason is that parathion and malathion are converted into paraxon and malaxon. This conversion is generally termed as oxidation, but since there is no change in valency, the appropriate term for this reaction would be desulfuration or bioactivation. In the body, sulfur atom is the molecule of parathion and malathion. This molecule is replaced by oxygen; hence in place of P=S, the formation of P=O takes place. Since =O is more electrophilic than =S (i.e., it attracts more electrons), P in P=O compound acquires sufficient positive charge to interact rapidly with cholinesterase enzyme.

The conversion of malathion to malaxon is an example of isomerization of thiono to the thio type. This is the reason why phosphorothionates are latent or indirect inhibitors of cholinesterase enzyme and act after conversion to P=O form. The chemical isomerization is slow at normal temperature but it may be enzymatically catalyzed in the organism. Since organophosphorus insecticides are manufactured at elevated temperatures, phosphorothionate is likely to be contaminated with thio isomers which may cause direct inhibition of cholinesterase enzyme.

REACTION BETWEEN ACETYLCHOLINE AND CHOLINESTERASE

The reaction between acetylcholine and cholinesterase enzyme takes place in three stages, in which acetylcholine is hydrolyzed and the enzyme is recovered.

- *Stage I:* When acetylcholine reacts with cholinesterase, an enzyme complex, commonly known as “Michaelis complex,” is formed.
- *Stage II:* The enzyme complex yields choline and acetylated enzyme. This reaction is called acetylation reaction.
- *Stage III:* In the last stage, deacetylation reaction takes place in which acetylated enzyme is hydrolyzed to give free enzyme and acetic acid.




In living organisms, these reactions proceed fast so that there is no accumulation of acetylcholine across the synapse or neuromuscular junction. The choline portion is removed and the acetic acid combines again to form new acetylcholine, and the cycle is completed.

REACTION BETWEEN OP AND ChE ENZYME

OP compounds react with AchE in the same manner as the normal substrate acetylcholine. Therefore, the reaction between OP compounds and ChE enzyme is essentially analogous to the reaction between Ach and ChE enzyme in the early stages of the reaction, but in the last stages of the deacetylation, “Ach hydrolysis” occurs very rapidly and the enzyme is recovered, whereas dephosphorylation in the ChE inhibition reaction takes place at an extremely slow rate; as a result, the OP compounds are powerful ChE inhibitors.

The ChE enzyme has two active centers in its molecule, namely, anionic site and esteratic site. The esteratic site catalyses the hydrolysis of linkage, whereas the anionic site binds the trimethylammonium group and is negatively charged. The esteratic site contains three groups: basic (histidine imidazole), hydroxyl (serine), and acidic (tyrosine hydroxyl) (Fig. 1).

By the action of enzyme on paraxon, a reversible complex is formed. In this process, the hydroxyl group of the enzyme attacks the phosphorus atom of paraxon. The hydrogen atom of the acidic group is transferred to the  part of the paraxon to give *p*-nitrophenol. The remaining product is phosphorylated enzyme. The reaction is known as phosphorylation. The reaction is reversible but reversibility depends on two factors: i) affinity of the inhibiting compound and ii) rate of phosphorylation (Fig. 2).

Since the phosphorylation constant is very high, the reversible complex is immediately converted into phosphorylated enzyme which is called dephosphorylation. In normal deacetylation reaction of acetylated enzyme, the rate of deacetylation is very high (295,000 molecules/active center/min), whereas the rate of dephosphorylation is extremely slow (3 molecules/active center/min) which leads to extremely negligible amount of enzyme recovery.

In brief, the reaction may be described as given in and Scheme 3; where EH is the enzyme and AB is the insecticide in which A is the phosphorylating group and B is the leaving group. (The leaving group is the non-phosphoryl or non-carbamyl portion of organophosphorus or carbamate insecticides).

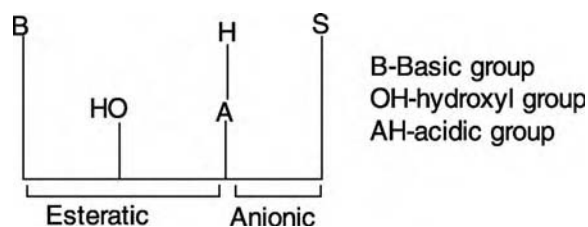


Fig. 1 Model of cholinesterase.

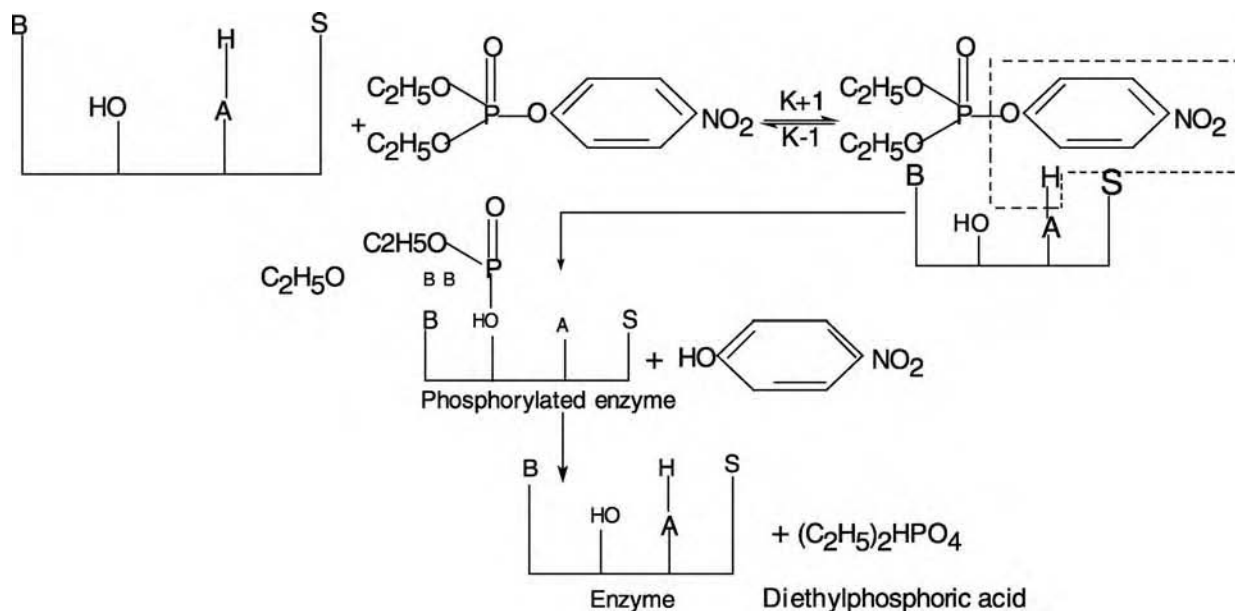
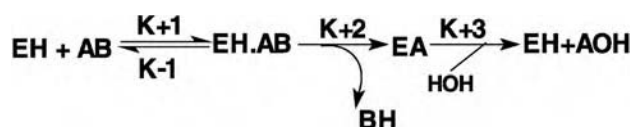


Fig. 2 Reaction of enzyme with paraxon.



Scheme 3

If the ChE enzyme is removed or a complex is formed from which the enzyme is released more slowly (as in the above reaction), the Ach will not be removed promptly from the receptor surface of the muscle. This would cause the muscle to remain depolarized longer than usual and will give rise to several action potential trials passing through the muscles resulting in the twitching of muscle, which leads to tetanus and eventually paralysis of the muscle.

REACTION BETWEEN CARBAMATE INSECTICIDE AND ChE ENZYME

Carbamates react with AchE in the same manner as the OP insecticides. Thus, the reason that the carbamates are toxic lies in their ability to inhibit the nerve enzyme AchE, resulting in the accumulation of Ach in the nerve synapse and causing a disruption of nerve function. The only difference is that the primary mode of ChE inhibition by carbamate is apparently reversible. It is due to the fact that in the carbamates, the second stage reaction is carbamylated and is slower than the OP; as a result, a small amount of enzyme is inhibited. However, it cannot be considered a general rule as there are carbamates that have higher values of

dissociation constant than OP compounds, and hence are good inhibitors of ChE.

CHLORINATED HYDROCARBONS

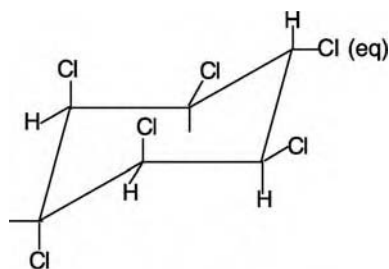
Although the use of DDT, especially in agriculture has been banned in the developed countries due to bioaccumulability (1970 in Sweden; 1971 in Japan; 1972 in U.S.A. and many other countries subsequently), it continues to be one of the most interesting insecticides ever discovered and is still being used to combat insect vectors of disease in some parts of the world such as India.

For the DDT group, no biochemical mechanism has been established, although the inhibition of the ATP-dependent portion of the Na/Ca exchange may be involved.^[5] It is now well established that DDT acts primarily on neurons and interferes with the axonal and synaptic transmissions. DDT prolongs the inward sodium current (delay of sodium channel closing; the prolongation of falling phase) and increases the depolarization after potential. When the depolarization after the potential is increased to a certain level, a sudden burst of repetitive discharge and a train of impulses can be provoked by a single stimulus. This leads to hyperexcitability of the nervous system, resulting in tremor, paralysis, and eventually death of the insects.

HEXACHLOROCYCLOHEXANE (HCH, BHC)

Hexachlorocyclohexane (previously called BHC, benzenehexachloride) is the oldest of the OC insecticides.

There are theoretically many isomers of HCH, in which seven (α , β , γ , δ , ϵ , η , and θ) are known (Scheme 4).



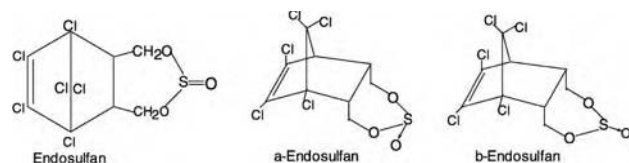
Gamma-HCH (lindane)

Scheme 4

The toxic principle in HCH is γ -isomer, named after its discoverer. Three chlorine substituents take axial (ax or a) and the other three take equatorial (eq or e) conformations on the chair form of cyclohexane. Lindane does not interact with any specific enzyme, although it is better inhibitor of Na, K, and Mg-ATPase than DDT. It causes an accumulation of Ach in the nerves of insects but it does not inhibit the enzyme ChE.

CYCLODIENES

Cyclodienes compounds are a group of highly active insecticides. However, many of them have long persistence and their use is restricted. They include chlordane, heptachlor, aldrin, dieldrin, isodrin, endrin, endosulfan, and mirex but endosulfan is the most commonly used compound, which have low persistence. It undergoes oxidation in insects, mammals, and plants to form a primary insecticidal metabolite, endosulfan sulfate. Endosulfan and the sulfate are hydrolyzed to form endosulfan diol (Scheme 5).



Scheme 5

Cyclodienes compounds act as neurotoxicants and the primary mode of action is blocking GABA-gated chloride channels like HCH, but not like DDT, and these compounds have positive temperature relationship (the opposite effect is seen in HCH and DDT).

They cause an excessive release of Ach at presynaptic sites, but they do not inhibit the enzyme ChE and do not affect any other enzyme system; however, there is evidence that they interact with ATPases from the nerve cord and muscle.^[6] The toxicities of heptachlor, aldrin, and isodrin are increased due to their conversion to their corresponding epoxides, heptachlor epoxide, dieldrin, and endrin. Dieldrin is the most persistent among the cyclodienes; it accumulates in fatty tissues and is retained for long periods of time. Endrin does not accumulate in fat and is metabolized to water soluble metabolites, which are excreted.

SYNTHETIC PYRETHROIDS

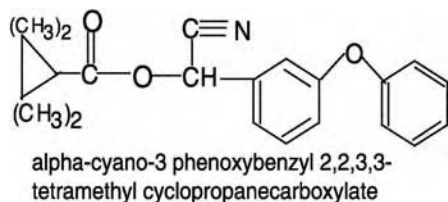
Pyrethroids are synthetic compounds based on natural pyrethrins as models, generally arrived at by systematic variations of parts for the purpose of improving photostability and insecticidal activity.^[7] All pyrethroids are lipophilic compounds, almost insoluble in water; in these respects, they resemble the OCs but differ from most OP and carbamates. There are four generations of pyrethroids:

- First generation: Allethrin (1949).
- Second generation: The pyrethroids included in this category are dimethrin (1961), tetramethrin (phthalthrin, 1965), resmethrin and bioremethrin (1969), and bioallethrin (1969).
- Third generation: The third generation pyrethroids comprise the most light-stable compounds which achieved wide application in agriculture. The first light-stable compound introduced was fenprothrin in 1971 but was commercialized as an acaricide in 1980. During this period, the most active and most stable compounds—permethrin, cypermethrin, deltamethrin, and fenvalerate—were introduced.
- Fourth generation: These pyrethroids were introduced during the period 1975–1983. Flucythrinate and flualinate were reported to be a broad spectrum insecticide with activity against phytophagous mite. Cyfluthrin was introduced in 1981 against cotton insects and later on broad spectrum cycloprothrin and fenpyrithrin insecticides.

Pyrethroids show negative temperature dependence of insect killing effect. In some cases, type II pyrethroids show positive temperature dependence; nerves respond to pyrethroids to produce repetitive discharges generally at low temperatures, but there is an optimal temperature range for this effect; the negative temperature is explainable in terms of more drastic changes of sodium currents at low temperatures.

USES OF PHOTOSTABLE PYRETHROIDS

The latest group of synthetic pyrethroids is photostable as well as extremely toxic to insects. Their efficacy is so good that a dose of 10–40 g active ingredient per hectare is required (Scheme 6).



Scheme 6

In 1976, OP compounds represented 40% of the world agricultural market, OC 30%, carbamate 15%, and miscellaneous 5%. In 1983, OP compounds represented 35–40%, OC 15%, carbamate 20%, pyrethroids 20–25%, and miscellaneous 5%. Of the pyrethroid insecticide market in 1983, permethrin represented 3%. These broad spectrum photostable pyrethroids are effective against a wide variety of insect pests, harmless to mammals and birds, and not phytotoxic. They combine high insecticidal activity with suitable persistence (they are as much as 10-folds more effective in the field than the most potent compounds of the other three principal groups); they show high toxicity toward lepidopterous larvae on many crops, especially *Heliothis* and *Spodoptera* species on cotton and against insect pests of forests.^[8–10]

NEONICOTINOIDS AND NITROGENOUS

Imidacloprid [1-(6-chloro-3-pyridylmethyl)-N-nitroimidazolidin-2-ylideneamine], a novel insecticide, derived synthetically from a nitromethylene insecticidal chemical, nithiazine, is closely related to nicotinoids in mode of action and structure activity relationship. Later on, several insecticides resembling imidacloprid appeared, which are collectively called neonicotinoids. These insecticides interact with the Ach binding site of the nicotinic Ach receptor as agonists, which causes excitation and eventually paralysis leading to death, as nicotine does.

HERBICIDES

Herbicides have provided a more effective and economical means of weed control than mechanical cultivation. Together with fertilizers and improved variety

of plants, herbicides have made an immense contribution to increasing crop yield with reduced costs. There are a wide variety of chemical compound types in use as herbicides. In general, the herbicides are less of a toxic hazard to humans and domestic animals than do the compounds used as insecticides. However, some herbicides, such as 2,4,5-T and silvex, possess toxic potential to humans based on contaminants.

Chlorophenoxy Acids

The chlorophenoxy acid-type herbicide includes 2,4-D (2,4-dichlorophenoxyacetic acid), 2,4,5-T (2,4,5-trichlorophenoxyacetic acid), and silvex [2-(2,4,5-trichlorophenoxyacetic) propionic acids]. These compounds are formulated in the form of various salts and esters to produce materials of low volatility in order to minimize the risk to useful plants growing near treated fields. Compounds in this class exert their toxic action on plants by acting as growth hormones. They apparently have no other hormonal action in plants.

Carbamate Herbicides

Carbamate herbicides are esters of carbamic acids and exhibit fungicidal as well as herbicidal activity but differ from carbamate insecticides in having little or no AchE activity. Among the commonly used herbicides of this group such as propham (isopropyl-N-phenylcarbamate) and chloroprotham, interferes with the photosynthetic activity. These herbicides are absorbed by plant roots and translated in the xylem.

Triazines and Triazoles

These heterocyclic cyclic nitrogen compounds have a low-order acute mammalian toxicity. They have the carcinogenic effect of the triazole compound amitrole (3-amino-1,2,4-triazole). Triazole herbicides include atrazine (2-chloro-4-ethylamino-6-isopropylamine-S-triazine) and simazine [2-chloro-4,6-bis(ethylamino)-S-triazine]. These are persistent soil-acting herbicides, which can be applied in large concentrations as total weed killers on rights-of-way. Triazines act by interfering with photosynthesis, and their primary site of action is inhibition of the Hill reaction of photosynthetic electron transport.

FUNGICIDES

Fungicides, like insecticides and herbicides, are comprised of a heterogeneous group of compounds, many of which are chemically unrelated.

Dithiocarbamates

Dithiocarbamates are derivatives of sulfur-containing dithiocarbamic acid. The chemical structures involved in dimethyldithiocarbamates are in combination with metallic salts such as zinc salt (ziram), ferric salt (ferbam) and manganous salt (maneb); these compounds offer a particular affectivity, better stability, and less phytotoxicity than elemental sulfur. Their toxic effects to fungi probably stems from isothiocyanate-radical ($-N=C=S-$), which is formed as a breakdown component. Additionally, helates are formed within the fungal cells when dithiocarbamates or heavy metal fungicides are applied. When excess quantities of such chelates are present, they may interfere with the enzymic and metabolic process within the cells. Heavy metal dithiocarbamates have great killing power. Mancozeb [a complex of zinc (2–5% Zn) and maneb (20%)] is ready-to-use fungicide that combines the benefit of both maneb and zineb.

Organotin compounds

The most widely used organotin fungicides are triphenyl tin (fentin) salts such as fentin acetate, fentin hydroxide, and fentin chloride.

Dicarboximides

Dicarboximides are also known as sulfenimides as they contain sulfur and nitrogen atom at the central position. Dicarboximides are considered to be among the safest fungicides and are used as seed treatment and for protectant spray for *Sclerotinia* diseases.

Oxathiins

Carboxin, furmecyclox, methfuroxam, oxycarboxam, and other related compounds are mainly effective against basidiomycetes, which are a class of fungi that includes such important diseases as smuts and rusts of cereals.

Benzimidazoles and thiophanates

This group also includes highly effective, systemic, broad-spectrum fungicides such as benomyl, carbendazim, thiobendazole, and thiophanate-methyl. Fuberizol is an important replacement for organomercury compounds as seed dresser.

Ergosetrol biosynthesis inhibitors (EBIs)

EBIs are a chemically heterogeneous group of systemic fungicides, grouped together because of a similar mode of action. They are also called sterol biosynthesis-inhibiting

fungicides (SBIs) or demethylation inhibitors (DMIs). Among the imidazoles imazalil, procloraz, fenapanil, and in the piperazine, pyridine, and pyridine compounds, fenarimol, pyrifenoxy, and triforine are important inhibitors, while in the morpholins group, aldimorph, tridemorph, and dodemorph demonstrate a systemic, specific activity against powdery mildews of cereals and ornamentals. Bitertanol, myclobutanil, and flusilazol are important in the triazole group.

PESTICIDES AS PERSISTENT ORGANIC POLLUTANTS AND ALTERNATIVE

Pesticides in general are an indispensable part of modern farm practices and have enabled us to obtain new standards of food production and quality. Pesticides are not an ecological sin if their use is restricted to judicious use and with common sense. People exposed to some highly toxic compounds may suffer short-term or long-term health problems. Excessive residues in the environment may contaminate water supplies and lead to lower water quality. They may contaminate our food through residues on sprayed crops. Pesticides may cause injury to non-target organisms such as bees, bird, other wildlife, and natural enemies of pest insects. Improperly applied pesticides may cause damage to treated surfaces, or through drift to surfaces adjacent to treated areas. Some pesticides may be phytotoxic, i.e., injurious to crops and ornamental plants.

It is also certain that pesticides will continue to be used for a considerable period of time in the future, but the hazards of pesticide chemicals need to be culled out. The future pesticides will have high potency chemicals requiring less dosage per unit area of effectiveness. This trend is already apparent from recently developed pyrethroids. Consumer groups and the general public may also be able to support the implementation of integrated pest management (IPM) programs by demanding residue-free commodities. There is now a distinct market for organically produced food and other products. Non-Governmental Organizations (NGOs) and consumer groups need to be strengthened, especially in developing countries, so that there will be public-oriented movements that will push for the implementation of IPM. IPM takes a systematic view of crop production to manage crop systems and employ pest-control tactics including biological and cultural alternatives, biorational pesticides, and judicious use of conventional pesticides. Further, IPM does not mean the absence of chemical control. Technological advancements in pest control will continue to be incorporated.

From the foregoing account, it is clear that there is a vast array of possibilities and opportunities as alternatives to pesticides to combat the various ills associated with pesticides use, but the selection of the right

chemicals is the wisest option. The future thus belongs to IPM strategy.

REFERENCES

1. Perry, A.S.; Yamamoto, I.; Ishaaya, Perry, R.Y. *Insecticides in Agriculture and Environment: Retrospects and Prospects*; Springer-Verlag: Berlin, 1999; 1–261.
2. Osteen, C.D.; Szmedra, P.I. *Agricultural Pesticide Use Trends and Policy Issues*; USDA Economic Research Service, 1989; 622 pp.
3. Anonymous strategy for developing the pesticide industry or news in brief. *Pesticide Information* **1999**, 25 (2), 50–52.
4. Martin, H. *Pesticide Manual*, 3rd Ed.; British Crop Protection Council, 1972.
5. Matsumura, F.; Clark, J.M. ATP-utilizing system in the squid axons: a review on the biochemical aspects of ion-transport. *Prog. Neurobiol.* **1982**, 18, 231–255.
6. Matsumura, F. *Toxicology of Insecticides*, 2nd Ed.; Plenum Press: New York, 1985; 1–598.
7. Davies, J.H. The pyrethroids: a historical introduction. In *The Pyrethroid Insecticides*; Leahey, J.P., Ed.; Taylor and Francis: London, 1985; 1–41.
8. Elliott, M. Synthetic Pyrethroids. In *Synthetic Pyrethroids*; Elliott, M., Ed.; ACS Symposium Series; American Chemical Society: Washington, DC, 1977; 42, 1–28.
9. Elliott, M.; Janes, N.F.; Potter, C. The future of pyrethroids in insect control. *Annu. Rev. Entomol.* **1978**, 23, 443–469.
10. Herve, J.J. Agricultural and public health usage. In *The Pyrethroid Insecticides*; Leahey, J.P., Ed.; Taylor and Francis: London, 1985; 343–417.

Cherry Diseases: Ecology and Control

George W. Sundin

Department of Plant Pathology, Michigan State University, East Lansing, Michigan, U.S.A.

INTRODUCTION

Sweet cherry (*Prunus avium* L.) and tart cherry (*Prunus cerasus* L.) trees are subjected to numerous fungal, bacterial, and viral diseases that limit the production of high quality fruit in commercial orchards. These diseases either directly impact fruit production by causing rotting or fruit spot symptoms or impact tree health through canker formation or by inciting premature defoliation. The major cherry diseases that will be discussed in this entry are the fungal diseases brown rot, leaf spot, and powdery mildew and the bacterial disease bacterial canker. This article discusses aspects of the ecology of these diseases individually and summarize with information on current control practices. Most cherry pathogens are active in spring and early summer and later in the fall except for the leaf spot fungus, which is active throughout the growing season. Because of the lack of effective host resistance, control of most cherry diseases is currently accomplished through the application of bactericides or fungicides.

BROWN ROT

American brown rot, caused by the fungus *Monolinia fructicola*, is a common stone fruit disease that also affects apricot, peach, nectarine, and plum. Brown rot is most prevalent on sweet cherry but is also economically important on tart cherry. The brown rot fungus attacks blossoms, spurs, and shoots, and causes the most conspicuous symptoms on fruit, which can be colonized and decayed within 24 hr under favorable conditions—warm and humid weather (Fig. 1). Fungal spores (conidia) are produced in abundance on infected fruit, furthering pathogen spread. Infected fruit persist as mummies on trees or the ground; *M. fructicola* overwinters in these mummies producing sexual fruiting bodies (apothecia) the following spring. Ascospores produced within apothecia on the ground are forcibly ejected during bloom and carried by wind to blossoms where infection occurs.

LEAF SPOT

Symptoms of cherry leaf spot, caused by the fungus *Blumeriella jaapii* (formerly known as *Coccomyces*

hiemalis), are most prevalent in late summer and appear as small purple circular spots that turn brown as the lesions age (Fig. 2A). Conidia produced in lesions are visible on the underside of leaves as white spore masses; the spores are disseminated by rain and wind (Fig. 2B). Leaves that have accumulated a sufficient number of lesions turn yellow and abscise, resulting in premature defoliation of trees. Trees that have defoliated prematurely cannot produce and transport enough photosynthate to root systems and become highly susceptible to and can be killed by winter injury. In addition, leaf spot-defoliated trees that survive the winter emerge with reduced reserves the following season and exhibit reduced fruit set.

Cherry leaf spot is similar to the disease apple scab in that the fungal pathogen overwinters in diseased leaves on the orchard floor. In spring, apothecia are produced in these leaves, and ascospores are released following a wetting event. Temperature and the length of the wetting period are the factors controlling the primary infection by ascospores, with optimum infection occurring at temperatures of 15.5–20°C.^[1] In disease epidemic years, primary leaf spot infections occur on leaves and also on fruit and fruit pedicels, while in typical years, primary symptoms are less apparent. The secondary cycles of cherry leaf spot initiated by conidia are responsible for the majority of symptoms seen yearly in tart cherry orchards in the Northern U.S.A.

POWDERY MILDEW

Powdery mildew, caused by the fungus *Podosphaera oxycanthae*, is an occasional problem in most cherry-producing regions but can be a severe problem in arid fruit-producing areas such as the Pacific Northwest U.S.A. Powdery mildew is characterized by the production of a white fungal mat on the surfaces of leaves (Fig. 3), and fruit symptoms can also occur. Powdery mildew disease typically occurs under dry conditions, and mildew fungi are exceptional in that spore germination can sometimes occur in the absence of free water. Powdery mildew can reduce yield directly by colonizing fruit and indirectly by reducing photosynthesis because of fungal growth covering leaves.



Fig. 1 Brown rot symptoms on “Montmorency” tart cherry fruit showing fungal colonization and sporulation on fruit and developing mummies.

BACTERIAL CANKER

Bacterial canker [causal agents *P. syringae* pvs. *syringae* and *morsprunorum* (*Pss* and *Psm*)] is the most structurally destructive disease of sweet cherry, killing buds, fruiting spurs, and even entire branches and young trees. Cankers can rapidly girdle limbs on younger trees and are often accompanied by gummosis, believed to be a defense response by the tree in an attempt to restrict pathogen growth (Fig. 4). Bacterial canker is especially devastating to sweet cherry, and to a lesser extent tart cherry, following spring frost injury. The infection of spurs and subsequent canker formation is typically preceded by blossom infection. *P. syringae* is an opportunistic pathogen in that infection only occurs on hosts that are predisposed to infection. Stressed trees, for

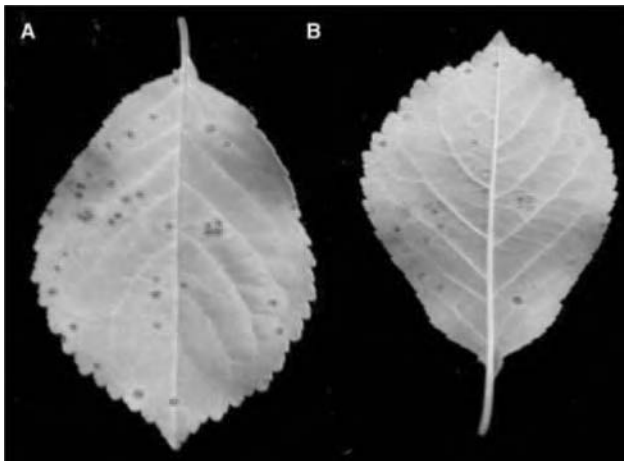


Fig. 2 Cherry leaf spot symptoms on (A) upper and (B) lower leaf surfaces of “Montmorency” tart cherry. White masses of conidia are apparent on the underside of leaves.



Fig. 3 Powdery mildew symptoms on “Montmorency” tart cherry leaves.

example, those grown on poor or marginal soils, can also be predisposed to canker infection.

Disease symptoms most commonly occur during bloom and after freeze events or after prolonged periods of cool, wet weather during bloom. Several physiological features of sweet cherry trees and the *Pss* pathogen contribute to the interaction of frost injury and bacterial canker infection. Sweet cherry blossoms and developing green tissue immediately preceding bloom are highly susceptible to frost injury. Frost-damaged tissue provides entry points for *Pss*, and water congestion in thawing tissues can rapidly move interior *Pss* populations within the tree.^[2] The *Pss* bacterium also directly contributes to frost injury because many strains of *Pss* are ice nucleation active and provide biological ice nuclei that can catalyze ice



Fig. 4 Cankers and killed fruiting spurs on scaffold limbs of sweet cherry caused by the bacterial canker pathogen *Pss*.

formation.^[3] The severity of bacterial canker symptoms increases with decreasing freezing temperatures and duration of freezing periods experienced by trees. For example, mild frosts [-2.2 to -0.6°C (28 – 31°F)] of relatively short duration (2–5 hr) may only result in infections that kill flower pistils but leave blossoms intact (G.W. Sundin, unpublished information). More severe frosts ($<-2.2^{\circ}\text{C}$) usually result in blossom blast, a symptom in which petals are blackened and the flower is killed. Severe frosts of long duration, such as a frost that occurred in northern Michigan in 2002 [-3.3°C (26°F), 11 hr duration], result in blossom blast and accompanying wood invasion and canker formation. The dependence of bacterial canker infection on frost events is probably responsible for the sporadic occurrence of this disease in most regions.

Bacterial canker symptoms also include necrotic leaf spots that are surrounded by chlorotic yellow halos; these leaf spots tend to fall out of leaves as they age, and are termed “shot-holes.” Circular lesions on immature fruit also occur but tend to only be important on specific varieties. Both *Pss* and *Psm* can survive and maintain populations on symptomless leaves of sweet and tart cherry throughout the summer months, and these pathogens will recolonize trees through leaf scars left by dropping leaves in the fall.^[4]

VIRUS DISEASES

Cherries can be infected by one or more of a variety of viruses of which the Ilarviruses, Prunus necrotic ring-spot virus (PNRSV) and Prune dwarf virus (PDV), are particularly important.^[5] Depending on the strain, PNRSV infection can markedly reduce tart cherry yields. PDV infection results in a disease termed sour cherry yellows, the most important effect of which is a long-term decline in tree vigor and productivity. The interaction of infecting viruses can also be important as trees dually infected with Cherry leaf roll virus and PNRSV are subject to a rapid decline.

MANAGEMENT OF CHERRY DISEASES

There are few reports of host resistance to the main diseases of sweet and tart cherry; thus, the majority of control efforts for these diseases rely on bactericide and fungicide applications. In the North Central cherry-growing region of the U.S.A., fungal disease control strategies are initiated during bloom with applications targeted against brown rot blossom infection and primary leaf spot infection. The most important fungicides for brown rot control are iprodione, anilopyrimidines, sterol inhibitors (SIs) (e.g., fenarimol, myclobutanil, tebuconazole), and strobilurins.

After bloom, brown rot fungicide applications are designed to protect fruit from infection and are most often utilized within a 21-day time period prior to harvest. The most important fungicides for leaf spot control are chlorothalonil, captan, SIs, and strobilurins, and powdery mildew is targeted by sprays of SIs or strobilurins. Because of residue concerns, chlorothalonil may not be applied to fruit after “shuck split,” which is about one week after petal fall, although it can be used again after harvest. Leaf spot fungicide applications continue through harvest with additional applications after harvest designed to prolong the maintenance of healthy leaves on trees.

A significant problem with the use of fungicides with single target sites in plant disease control is the development of fungicide resistance. Resistance to SIs and strobilurin fungicides has been documented in many pathogens,^[6] and we have isolated *B. jaapii* isolates with resistance to SI fungicides from orchards in Michigan (Proffer et al., unpublished information). Because of the lack of availability of an extensive number of fungicide chemistries for disease control, the impact of fungicide resistance on the cherry industry is predicted to be dramatic.

Management of bacterial canker is exceedingly difficult on susceptible varieties because of the lack of control options. Copper is the only registered bactericide in many regions for bacterial canker control; unfortunately, sweet cherry trees are also highly susceptible to copper phytotoxicity. High rate copper sprays are typically only applied while trees are dormant, a timing when *Pss* and *Psm* populations are inaccessible, harbored within dormant buds. Lower rate (25–35% of high rate) copper applications are generally safe if made between bud break and the green tip stage but are usually discouraged during bloom. Thus, even though sprays are most needed for control of *Pss* populations on blossoms, they typically are not used because of phytotoxicity concerns. Copper resistance has also been detected in *Pss* bacterial canker strains, and resistance can further reduce the efficacy of copper for disease control.

Control or reduction of the effect of the Ilarviruses PNRSV and PDV is accomplished through the use of clean, certified planting material. However, in addition to being graft transmissible, these viruses are also pollen transmitted, and the western flower thrips is thought to be a key vector. Thus, control of thrips populations and control of broadleaf weeds in orchards during bloom are other methods to reduce virus spread.

CONCLUSIONS

The almost universal usage of the cultivar “Montmorency” in the tart cherry industry presents a disease

control nightmare, as this variety is highly susceptible to leaf spot, and susceptible to brown rot and powdery mildew. There has been little progress made in identifying potential biological controls for cherry diseases, with the most difficult issue being the requirement for almost season-long protection of a variety of host tissue from distinct pathogens. As such, increased research is needed on cherry disease control, particularly in the area of host resistance to diseases and the development of novel, efficacious, and reduced-risk fungicides and bactericides.

REFERENCES

1. Eisensmith, S.P.; Jones, A.L. A model for detecting infection periods of *Coccomyces hiemalis* on sour cherry. *Phytopathology* **1981**, *71*, 728–732.
2. Vigoroux, A. Ingress and spread of *Pseudomonas* in stems of peach and apricot promoted by frost-related water-soaking of tissues. *Plant Dis.* **1989**, *73*, 854–855.
3. Lindow, S.E.; Hirano, S.S.; Barchet, W.R.; Arny, D.C.; Upper, C.D. Relationship between ice nucleation frequency of bacteria and frost injury. *Plant Physiol.* **1982**, *70*, 1090–1093.
4. Sundin, G.W.; Olson, B.D.; Jones, A.L. Overwintering and population dynamics of *Pseudomonas syringae* pv. *syringae* and *P. s.* pv. *morsprunorum* on sweet and sour cherry trees. *Can. J. Plant Pathol.* **1988**, *10*, 281–288.
5. Uyemoto, J.K.; Scott, S.W. Important diseases of *Prunus* caused by viruses and other graft-transmissible pathogens in California and South Carolina. *Plant Dis.* **1992**, *76*, 5–11.
6. Gisi, U.; Chin, K.M.; Knapova, G.; Farber, R.K.; Mohr, U.; Parisi, S.; Sierotzki, H.; Steinfeld, U. Recent developments in elucidating modes of resistance to phenylamide, DMI and strobilurin fungicides. *Crop Prot.* **2000**, *19*, 863–872.

Cherry Insects: Ecology and Control

Helmut Riedl

Mid-Columbia Agricultural Research and Extension Center, Oregon State University,
Hood River, Oregon, U.S.A.

Jesus Avilla

Centre UdL-IRTA for R+D, University of Lleida, Lleida, Catalonia, Spain

INTRODUCTION

Two species of cherry are grown for their fruit: sweet cherry, *Prunus avium* L., and sour cherry, *Prunus cerasus* L. Both species are native to Southeastern Europe and Western Asia between the Black Sea and the Caspian Sea.^[1] Cherry production and many of the important cultivars originated in Europe and spread from there to other parts of the world. Most sweet cherry cultivars are grown for the fresh-fruit market, whereas sour cherries are processed for various food uses. Cherries are grown throughout the world. However, sweet cherry production is primarily found in regions with little or no rain during late spring and early summer due to the susceptibility of the fruit to rain cracking. The Food & Agriculture Organization (FAO) lists 65 countries where cherries are grown in commercial quantities.^[2] Total cherry acreage worldwide is 401,401 ha (Table 1). In terms of harvested acreage, Europe leads production with about 260,000 ha of cherries, followed by Asia (including Asia Minor) with 94,200 ha, North America with 32,633 ha, South America with 9358 ha, Africa with 3326 ha, and Australia and New Zealand with a combined 1950 ha (Table 1). In the United States, sweet cherry production is concentrated in the three western states of California, Washington, and Oregon, and Michigan, New York, and Utah are major producers of sour cherries. For the purposes of this review, sweet and sour cherries are treated as one commodity because they have a similar pest complex and comparable control programs.

ARTHROPOD PESTS OF CHERRIES AROUND THE WORLD

Cherries are attacked by a large number of arthropod pests. Table 2 is not a complete checklist but lists pests presently considered problems by plant protection specialists in their respective cherry districts, as well as those that appear in official pest control recommendations. For more detailed information about cherry pests in different geographic regions of the world, the

following sources can be consulted: North America,^[3,4] Europe,^[5] Turkey,^[6] and New Zealand.^[7] Larvae of several scarabaeids feed on the roots of cherries and other fruit trees, whereas the adults feed on flowers, buds, and leaves (Table 2). The buprestid *Capnodis tenebrionis* L. is a major root pest of cherries and other stone fruits in semiarid areas of southern Europe, around the Mediterranean, and Asia Minor. The related buprestid *C. carbonaria* L. also attacks the roots of stone fruits but occurs primarily in the eastern Mediterranean, Asia Minor, and the area between the Caspian and Black Sea.^[8] Larvae of the California prionus, a cerambycid, attack the roots and crown area of cherry trees in Utah. Cambium-feeding and wood-boring scolytid beetles, native to Europe and collectively known as shothole borers, attack cherries and are now widely distributed throughout the world (Table 2). Wood-boring Lepidoptera occasionally do serious damage to cherry trees. The cherry bark tortrix has a wide geographic range in Europe, as do two other lepidopteran pests: the leopard moth and goat moth. The cherry bark tortrix has recently been introduced into the Pacific Northwest of the United States but has not yet spread into commercial cherry orchards. The leopard moth is present in the eastern United States but has not yet developed into an orchard problem there, though it was recently detected in Ontario, Canada.^[9] The American plum borer, a cambium feeder native to North America, has adopted cultivated cherries as a host, but it is not a problem in the Pacific Northwest. The lesser peach tree borer infests the cambium tissues of the upper trunk and the scaffold branches of cherries and other stone fruit trees in North America. Larvae of the peach tree borer damage the cambium tissues at the base of the trunk at soil level and can girdle young cherry trees. Several species of scales can also infest cherries. San Jose scale is the most serious and widespread scale pest. The mulberry scale is gaining importance in some Mediterranean areas (Table 2). Generally, mealybugs are minor pests on cherries except for the apple mealybug in British Columbia, which vectors a virus causing "little cherry disease."^[10] In Oregon, pear thrips can destroy buds

Table 1 Harvested cherry area (in hectares) for countries reporting commercial cherry production; 2005 data available at FAOSTAT

| Country/continent | Total cherry area in hectares by country/continent |
|---------------------------|--|
| Europe | |
| Spain | 49,112 |
| Germany | 33,000 |
| Italy | 30,303 |
| Russian Federation | 30,000 |
| Ukraine | 14,000 |
| France | 13,000 |
| Poland | 10,300 |
| Bulgaria | 10,150 |
| Serbia and Montenegro | 10,000 |
| Greece | 9,500 |
| Romania | 9,317 |
| Portugal | 6,250 |
| Belarus | 4,000 |
| Georgia | 4,000 |
| Switzerland | 4,000 |
| Austria | 3,500 |
| Bosnia and Herzegovina | 3,000 |
| Moldova, Republic of | 3,000 |
| Hungary | 2,500 |
| Croatia | 2,000 |
| Belgium | 1,277 |
| Macedonia | 1,200 |
| Latvia | 1,100 |
| Czech Republic | 1,090 |
| Albania | 1,000 |
| Estonia | 800 |
| Lithuania | 687 |
| United Kingdom | 380 |
| Netherlands | 300 |
| Norway | 300 |
| Cyprus | 265 |
| Sweden | 190 |
| Slovakia | 130 |
| Slovenia | 113 |
| Luxembourg | 100 |
| Denmark | 70 |
| Europe (total) | 259,934 |
| Asia | |
| Turkey | 26,000 |
| Iran, Islamic Republic of | 25,700 |
| Syrian Arab Republic | 10,000 |
| Lebanon | 7,600 |
| China | 4,500 |
| Uzbekistan | 4,500 |
| Japan | 4,260 |
| Kazakhstan | 4,000 |
| Kyrgyzstan | 2,000 |
| India | 1,700 |
| Pakistan | 1,200 |
| Armenia | 1,000 |
| Azerbaijan, Republic of | 1,000 |

(Continued)

Table 1 Harvested cherry area (in hectares) for countries reporting commercial cherry production; 2005 data available at FAOSTAT (Continued)

| Country/continent | Total cherry area in hectares by country/continent |
|--------------------------------|--|
| Israel | 400 |
| Jordan | 190 |
| Palestine—Occupied Territories | 150 |
| Asia (total) | 94,200 |
| Africa | |
| Algeria | 1,900 |
| Morocco | 1,270 |
| South Africa | 156 |
| Africa (total) | 3,326 |
| North America | |
| United States of America | 31,500 |
| Canada | 1,133 |
| North America (total) | 32,633 |
| South America | |
| Chile | 7,300 |
| Argentina | 1,333 |
| Bolivia | 330 |
| Guyana | 230 |
| Peru | 90 |
| Mexico | 75 |
| South America (total) | 9,358 |
| Australasia | |
| Australia | 1,400 |
| New Zealand | 550 |
| Australasia (total) | 1,950 |
| World (total) | 401,401 |

Source: From Ref.^[2].

and flowers on cherries adjacent to native hosts, such as maple. Occasionally, climbing cutworms are a problem in some North American cherry-growing areas, feeding on buds and new leaf tissue on lower branches just before bloom. In Utah, New York, and other states, fruitworms cause localized defoliation and bud and fruit damage after bloom. In Europe, the cherry fruit moth destroys buds and flowers. Shoot tips are sometimes damaged in California by European earwig and peach twig borer feeding. The largest number of arthropods attacks the foliage of cherries, including several mite species: various leafrollers; other lepidopterous larvae, such as the red-humped caterpillar; leafminers; black cherry aphid; leafhoppers; lace bugs; and cherry slug (Table 2). Black cherry aphid is a key pest in some European areas, New Zealand, and Chile. However, the most important pests are those that feed directly on the fruit. This group includes cherry fruit flies (CFF); *Rhagoletis cingulata* (Loew) in the eastern

Table 2 Cherry insect and mite pests in selected production areas around the world

| | | Pests in cherry production areas around the world ^a | | | | | | | | | | | | | | | |
|---------------------------|--|--|-------------------------|----|----|--------|----|----|--------|----|--------|-------|--------|-------|--------|-------------|----|
| Plant part attacked | Order and scientific name | Common name | North and South America | | | | | | | | | | | Asia | | Australasia | |
| | | | USA | | | Canada | | | Europe | | | | | Italy | Turkey | AUS | NZ |
| | | | NY | CA | OR | UT | WA | BC | D | ES | France | Italy | Turkey | AUS | NZ | | |
| Roots, crown area | Coleoptera | | | | | | | | | | | | | | | | |
| | <i>Capnodis tenebrionis</i> L. | Peach flatheaded rootborer | | | | | | | | | x | x | | | x | | |
| | <i>Capnodis carbonaria</i> L. | Almond flatheaded rootborer | | | | | | | | | | | | | x | | |
| | <i>Melolontha melolontha</i> (Linnaeus) | Common cockchafer | | | | | | | | | x | x | | | x | | |
| | <i>Pleocoma</i> spp. (various species) | Rain beetles | | | | | | | | | | | | | | | |
| | <i>Polyphylla decemlineata</i> (Say) | Tenlined June beetle | | | | | | | | | | | | | | | |
| | <i>Polyphylla turkmenoglu</i> (Petr.) | | | | | | | | | | | | | | | | |
| | <i>Prionus californicus</i> Motschulsky | California prionus | | | | | | | | | | | | | x | | |
| Wood or cambium | Coleoptera | | | | | | | | | | | | | | | | |
| | <i>Chrysobothris mali</i> Horn | Pacific flatheaded woodborer | | | | | | | | | | | | | | | |
| | <i>Scolytus rugulosus</i> (Müller) | Fruittree bark beetle | | | | | | | | | | | | | | | |
| | <i>Xyleborus dispar</i> (Fabricius) | European shothole borer | | | | | | | | | | | | | | | |
| | <i>Xyleborus saxaseni</i> (Ratzeburg) | Lesser shothole borer | | | | | | | | | | | | | | | |
| | Lepidoptera | | | | | | | | | | | | | | | | |
| | <i>Enarmonia formosana</i> (Scopoli) | Cherry bark tortrix | | | | | | | | | | | | | | | |
| | <i>Cossus cossus</i> (Linnaeus) | Goat moth | | | | | | | | | | | | | | | |
| | <i>Euzophera semifuneralis</i> (Walker) | American plum borer | | | | | | | | | | | | | | | |
| | <i>Synanthedon exitiosa</i> (Say) | Peach tree borer | | | | | | | | | | | | | | | |
| | <i>Synanthedon pictipes</i> (Grote and Robinson) | Lesser peach tree borer | | | | | | | | | | | | | | | |
| | <i>Zeuzera pyrina</i> (Linnaeus) | Leopard moth | | | | | | | | | | | | | | | |
| On bark (trunk, branches) | Homoptera | | | | | | | | | | | | | | | | |
| | <i>Parlatoria oleae</i> (Colvée) | Parlatoria scale | | | | | | | | | | | | | | | |
| | <i>Parthenolecanium corni</i> (Bouché) | European fruit lecanium | | | | | | | | | | | | | | | |
| | <i>Phenacoccus aceris</i> (Signoret) | Apple mealy bug | | | | | | | | | | | | | | | |
| | <i>Pseudolaucaspis pentagona</i> (Targioni-Tozzetti) | Mulberry scale | | | | | | | | | | | | | | | |
| | <i>Quadraspidionus ostreaeformis</i> (Curtis) | Oystershell scale | | | | | | | | | | | | | | | |
| | <i>Quadraspidionus perniciosus</i> (Comstock) | San Jose scale | | | | | | | | | | | | | | | |
| | <i>Saissetia oleae</i> (Olivier) | Black scale | | | | | | | | | | | | | | | |

(Continued)

Table 3 Traditional controls, recent registrations, and pesticides under development for key and secondary pests of cherries in the Pacific Northwest of the United States

| Pests | | Traditional controls | Recent registrations | Products under development |
|------------------------------------|-------------------------------|--|---|---|
| Scientific name | Common name | | | |
| <i>Rhagoletis indifferens</i> | Western cherry fruit fly | Malathion ^a Diazinon ^a | Spinosad ^b Imidacloprid | Other neonicotinoid insecticides (e.g., acetamiprid, thiacloprid, clothianidin) |
| <i>Rhagoletis fausta</i> | Black cherry fruit fly | Azinphosmethyl ^a Dimethoate ^a Carbaryl | Thiamethoxam | |
| <i>Choristoneura rosaceana</i> | Obliquebanded leafroller | Chlorpyrifos ^a | Spinosad ^b | |
| <i>Pandemis pyrusana</i> | Pandemis leafroller | <i>Bacillus thuringiensis</i> (Bt) | Methoxyfenozide Pyriproxyfen | |
| <i>Quadraspidiotus perniciosus</i> | San Jose scale | HMO (horticultural mineral oil) Chlorpyrifos ^a (pre-bloom) Diazinon ^a (pre- and post-bloom) | Pyriproxyfen (pre- and post-bloom) Imidacloprid (crawlers) | |
| <i>Frankliniella occidentalis</i> | Western flower thrips | Endosulfan | Spinosad ^b | |
| <i>Phyllonorycter blancardella</i> | Tentiform leafminer | | Spinosad ^b Methoxyfenozide Pyriproxyfen | |
| <i>Myzus cerasus</i> | Black cherry aphid | Endosulfan | Imidacloprid Thiamethoxam | Other neonicotinoid insecticides (e.g., acetamiprid, thiacloprid, clothianidin) |
| | Leafhoppers (several species) | Carbaryl Endosulfan | Imidacloprid Thiamethoxam | Other neonicotinoid insecticides (e.g., acetamiprid, thiacloprid, clothianidin) |
| <i>Scolytus rugulosus</i> | Fruittree bark beetle | Endosulfan Pyrethroids | | |
| <i>Xyleborus dispar</i> | European shothole borer | | | |
| <i>Schizura concinna</i> | Redhumped caterpillar | <i>Bacillus thuringiensis</i> (Bt) | Methoxyfenozide | |
| <i>Tetranychus urticae</i> | Twospotted spider mite | HMO Dicofol | Spirodiclofen | Etoxazole Bifenazate |
| <i>Panonychus ulmi</i> | European red mite | Fenbutatin-oxide Clofentezine Hexythiazox | | |

^aOrganophosphate (OP) insecticides.

^bDifferent formulations available for conventional and organic production.

United States; *R. indifferens* (Curran) in the western United States; and *R. fausta* (Osten Sacken), a less common but ubiquitous species found across North America. *Rhagoletis cerasi* (Linnaeus) is the ecological equivalent to the North American CFF species and is found in all major cherry-growing areas in Europe, as well as Turkey. Newer cherry-growing areas, such as Chile and New Zealand, are still free of CFF (Table 2). Occasionally, leafroller larvae cause fruit damage

and contaminate harvested cherries.^[3,11] Peach twig borer reportedly attacks the fruit in California. The plum curculio, a native pest of pome and stone fruits in eastern North America, has recently been detected on cherries in Utah.^[12] In Oregon and Washington, western flower thrips have been known to feed on the fruit surface close to harvest, especially on late-maturing cultivars, resulting in silvery, ringlike blemishes.^[11]

CONTROL OF CHERRY PESTS

Chemical Control

To achieve the high quality standards demanded by the market, cherry growers around the world have relied primarily on organophosphates (OP) and, to some extent, on other broad-spectrum chemistries (i.e., carbamates and pyrethroids) for control of CFF and other pests. In the United States and elsewhere, regulatory restrictions are forcing growers to reduce OP use in tree fruits and replace them with alternative controls. In recent years, OP alternatives and new control methods have become available, and they are beginning to change pest control practices for cherries, as illustrated by the situation in the Pacific Northwest (Table 3). Wherever CFF is part of the pest complex, it is the key pest and dominates the seasonal control program. In the United States, no CFF larvae are allowed in cherries delivered to a packinghouse. To meet that requirement, up to six insecticide sprays are applied annually for CFF control in Oregon.^[11] Among OP alternatives for CFF control are the neonicotinyl insecticides, imidacloprid and thiamethoxam, and various spinosad formulations. Two spinosad formulations are also approved for organic cherry production in the United States.^[13] Non-OP control alternatives are now also available for other pests (Table 3), including various Bt formulations against leafrollers; the insect growth regulators (IGRs) methoxyfenozide and pyriproxyfen against leafrollers and leafminers; spinosad against leafrollers, thrips, and leafminers; pyriproxyfen against various scale insects; and imidacloprid and thiamethoxam for control of black cherry aphid and leafhoppers. Current cherry pest management guidelines for different growing regions can be accessed at Web sites listed in this entry's references section: British Columbia,^[14] California,^[15] Italy,^[16] New York,^[17] Oregon,^[18] Spain,^[19] Turkey,^[6] Utah,^[20] and Washington.^[21]

BIOLOGICAL, BEHAVIORAL, AND CULTURAL CONTROL OF CHERRY PESTS

The frequent application of OPs and other broad-spectrum sprays for control of key cherry pests, in particular CFF, has been disruptive to natural enemies and has at times resulted in outbreaks of secondary pests.^[11] There is little opportunity for biological control of CFF, but other cherry pests are amenable to at least partial regulation by natural enemies. As cherry growers begin to shift from broad-spectrum insecticides to more selective controls, opportunities for biological control of spider mites, leafhoppers, leafminers, aphids, mealy bugs, and scale insects will

increase. Behavioral control methods, such as mating disruption with sex pheromones, have been developed for control of some cherry pests, including Oriental fruit moth, lesser peach tree borer, and leafrollers. A spinosad bait spray has recently been registered in the United States for control of CFF.^[11,13] Its advantages are speed of application, low spray volume per hectare, and selectivity to natural enemies due to selective placement. The effectiveness of this bait spray against the European CFF has not yet been demonstrated. Wood-boring insects—such as shothole borers, ambrosia beetles, and others—can be held in check by denying them breeding sites through sanitation measures. This involves maintaining trees in good health, and removing injured limbs and weakened or debilitated trees before they become infested. Insecticides are of only limited effectiveness.

REGULATORY CONTROL

In the Pacific Northwest of the United States, cherry orchards are part of CFF control districts. This means that all cherry orchards in an area have to be sprayed against CFF. Cherries delivered to packinghouses are regularly inspected for the presence of CFF or other insect larvae before they are accepted. Some markets, such as Japan and Taiwan, require that cherries be fumigated at the source with methyl bromide before they are shipped. According to the Montreal Protocol for the protection of the ozone layer, field uses of methyl bromide will be phased out.^[22] However, at this point, it appears that preshipment uses of methyl bromide will be maintained.

CONCLUSIONS

Pest management of cherries is undergoing major changes. Organophosphate insecticides, which comprised the principal control tools available to cherry growers for several decades, are under increased scrutiny and are slowly being phased out or, at the very least, becoming restricted in their use. Fortunately, alternative controls have become available for most cherry pests—controls that are as effective but not always as economical as the OP insecticides. In the western United States, organic sweet cherry production has been on the rise due to the availability of the natural insecticide spinosad for the control of major pests, including CFF, leafrollers, and thrips. Aerial application, which is still the method of choice for CFF control in the United States, is also becoming more and more controversial because of drift issues, especially close to residential areas and near surface water. The new spinosad bait

spray can be applied more efficiently than conventional ground-applied CFF sprays and may offer some hope as a potential replacement for aerial application. It is expected that biological control will play a larger role in the future as cherry growers adopt more selective control methods, especially for CFF control. Cherry production is also experiencing major horticultural changes with the introduction of new cultivars, size-controlling rootstocks, and training systems.^[23] These changes will also have consequences for pest management. For instance, late-season cultivars extend the growing season and will require additional sprays due to longer exposure of the fruit to pests. Many of the newer cherry orchards are planted on size-controlling rootstocks, which allow smaller tree sizes and higher tree densities per hectare. This will improve control, because good spray coverage is easier to achieve on smaller trees. It may also lead to potential savings in spray materials, because the canopy volume of high-density plantings is less than that of standard-size trees.

ACKNOWLEDGMENTS

The authors would like to thank the following individuals for providing information about cherry pests and their control in their respective growing districts: D. Alston, Utah; T. Vogel, Germany; H. Vogt, Germany; A. Ozdem, Turkey; O. Gurkan, Turkey; J. McLaren, New Zealand; M. Kulczewski, Chile; S. Caruso, Italy; and J. Dalmases, Spain. We also thank Dr. M. Willett, Northwest Horticultural Council, for reviewing the manuscript.

REFERENCES

1. Westwood, M.N. *Temperate Zone Pomology*; W.H. Freeman: San Francisco, CA, 1978.
2. Available at: <http://faostat.fao.org> (accessed July 2006).
3. Beers, E.H., Brunner, J.F., Willett, M.J., Warner, G.M., Eds.; *Orchard Pest Management: A Resource Book for the Pacific Northwest*; Good Fruit Grower: Yakima, WA, 1993.
4. Anonymous. Integrated pest management of stone fruits. Integrated Pest Management Project Pub, 3389, University of California: Berkeley, CA, 1999.
5. van Frankenhuisen, A.; Stigter, H. *Schädliche und nützliche Insekten und Milben an Kern-und Steinobst*; E. Ulmer: Stuttgart, Germany, 2002.
6. Available at: www.tagem.gov.tr/YAYINLAR/kiraz/icindekiler.htm (accessed July 2006).
7. McLaren, G.F.; Grandison, G.; Wood, G.A.; Tate, G.; Horner, I. *Summerfruit in New Zealand: Management of Pests and Diseases*; HortResearch, University of Otago Press: Dunedin, New Zealand, 1999.
8. Ben-Yehuda, S.; Assael, F.; Mendel, Z. Improved chemical control of *Capnodis tenebrionis* and *C. carbonaria* in stone-fruit plantations in Israel. *Phytoparasitica* **2000**, *28*, 1–16.
9. Available at: www.pestalert.org/viewArchPestAlert.cfm?rid=69 (accessed July 2006).
10. Available at: www.agf.gov.bc.ca/cropprot/tfipm/lcv.htm (accessed July 2006).
11. Riedl, H.; Walston, A.; Omeg, M.; Song, Y.; Castagnoli, S.; Long, L. Cherry pest management in transition: old and new pests, old and new pesticides. *Proceedings of Oregon Horticultural Society*, Vol. 94, Portland, Oregon, Jan., 27–29, 2004 Available at: www.oregonhorticulturalsociety.org (accessed July 2006).
12. Available at: <http://extension.usu.edu/cooperative/ipm/files/PDFDocs/PlumCurculio06.pdf> (accessed July 2006).
13. Available at: www.dowagro.com/usag/prod/index.htm (accessed July 2006).
14. Available at: www.agf.gov.bc.ca/treefruit/product/tfguide.htm (accessed July 2006).
15. Available at: <http://axp.ipm.ucdavis.edu/PMG/selectnewpest.cherries.html> (accessed July 2006).
16. Available at: www.ermesagricoltura.it/wcm/ermesagricoltura/consigli_tecnici/disciplinari/sezione_disciplinari/s_norme_coltura/s_frutticole/s_ciliegio/D_ciliegio.pdf (accessed July 2006).
17. Available at: www.nysaes.cornell.edu/ent/treefruit/pdf/2006TF12.pdf (accessed July 2006).
18. Available at: <http://extension.oregonstate.edu/catalog/pdf/em/em8203-e.pdf> and <http://extension.oregonstate.edu/catalog/html/em/em8329-e/> (accessed July 2006).
19. Available at: www.gencat.net/darp/c/camp/pi/doc/2005/normatec/nt05frpi.pdf (accessed July 2006).
20. Available at: http://extension.usu.edu/cooperative/ipm/files/publications/UCOIMMG_CAlston.pdf (accessed July 2006).
21. Available at: <http://cru.cahe.wsu.edu/CEPublications/eb0419/eb0419.pdf> (accessed July 2006).
22. Available at: www.unep.org/ozone/Treaties_and_Ratification/index.asp (accessed July 2006).
23. Whiting, M.D., Ed. *Producing Premium Cherries*; Good Fruit Grower: Yakima, WA, 2005.

Climate and Pest Outbreaks

Ana Iglesias

Goddard Institute for Space Studies, New York, New York, U.S.A.

Cynthia Rosenzweig

Goddard Institute for Space Studies, Columbia University, New York, New York, U.S.A.

INTRODUCTION

Climate affects not just agricultural crops but their associated pests as well. The spatial and temporal distribution and proliferation of insects, weeds, and pathogens is determined to a large extent by climate, because temperature, light, and water are major drivers for their growth and development. Climate also affects the pesticides used to control or prevent pest outbreaks (i.e., the intensity and timing of rainfall influence pesticide persistence and efficiency; and temperature and light affect pesticide persistence through chemical alteration). Most analyses concur that in a changing climate, pests may become even more active than they are currently, expanding their geographical range, and may engender increased use of agricultural chemicals that carry health, ecological and economic costs.

RESPONSE TO CLIMATE VARIABLES

Because of the extremely large variation of pest species' responses to meteorological conditions, it is difficult to draw overarching conclusions about the relationships between pests and weather. In general, however, most pest species are favored by warm and humid conditions. But crop damages by pests are a consequence of the complex ecological dynamics between two or more organisms and therefore are very difficult to predict. For example, dry conditions are unfavorable for sporulation of fungi, but are also unfavorable for the crop; a weak crop during a drought is more likely to become infected by fungi than when it is not stressed.

Precipitation—whether optimal, excessive, or insufficient—is probably the most important variable that affects crop–pest interactions. Both direct and indirect effects of moisture stress on crops make them more vulnerable and threaten damage by pests, especially in the early stages of plant development. Pest infestations often coincide with changes in climatic conditions, such as early or late rains, drought, or increases in humidity, which in themselves can reduce

yields. In these circumstances, attributing specific losses to pests can be difficult. Table 1 shows key weather conditions that critically influence major pest epidemics and examples of resulting crop damages.

Insects flourish in all climates, their habitats and survival strategies are strongly dependent on local weather patterns, and are particularly sensitive to temperature because they are cold-blooded. Insects respond to higher temperature with increased rates of development and with less time between generations. (Very high temperatures reduce insect longevity.) Warmer winters reduce winterkill, and consequently increase insect populations in subsequent growing seasons. Drought changes the physiology of host species, leading to changes in the insects that feed on them, and can reduce populations of friendly insects (like predators or parasitoids), spiders and birds, influencing the impact of pest infestations. Abnormally cool, wet conditions can also bring on severe insect and plant pathogen infestations, although excessive soil moisture may drown soil-residing insects.

Weeds compete with crops soil nutrients, light, and space. Drought conditions increase competition for soil moisture between crops and weeds, while humid conditions increase the proliferation of weeds. Warmer temperature regimes have been shown to increase the maximum biomass of grass weeds. Climate factors that influence the growth, spread, and survival of crop *pathogens* include temperature, precipitation, humidity, dew, radiation, wind speed, and circulation patterns. Increased temperature and humidity result in the spread of diseases, as wet vegetation promotes the germination of spores and the proliferation of fungi and bacteria, and influences the lifecycle of soil nematodes. Some pathogens (e.g., powdery mildews) thrive in hot, dry conditions as long as there is dew formation at night. Climate conditions also influence *post-harvest pest* damage. For example, the concentration of aflatoxin is raised during crop-water deficits, because drought favors the growth of the fungus producer *Aspergillus flavus* in the weakened crop. In contrast, mycotoxin (produced by *Fusarium* spp.) is favored by high humidity and temperature at harvest.

Table 1 Effect of weather events on pest damage and key observed examples*Floods and heavy rains*

- Increased moisture benefits epidemics and prevalence of leaf fungal pathogens
 - Rice leaf blight caused great famine in Bengal (1942), 2 million people died
 - Wheat stripe rust outbreak in major production regions of China contributed to the 1960s famine
 - Fungal epidemics in corn, soybean, alfalfa, and wheat (U.S. Midwest, 1993)
 - Mycotoxin (produced by *Fusarium* spp.) reached a record high (U.S. Great Plains, 1993); mycotoxin increases are related to high humidity during harvest (East Africa and South America, 1990s)
 - Humid summers drive epidemics of gray leaf spot of maize (Iowa and Illinois, 1996)
- Water induced soil transport increases dissemination of soilborne pathogens to non-infected areas
 - Outbreaks of soybean sudden death syndrome in the north central U.S. (1993)
- Continuous soil saturation causes long-term problems related to rot development and increase damage by pathogens
 - In maize, crazy top and common smut

Drought

- Water stress diminishes plant vigor and alters C/N lowering plant resistance to nematodes, and insects. Attack by fungal pathogens of stems and roots are favored by weakened plant conditions. Dry and warm conditions promote growth of insect vector populations, increasing viral epidemics
 - Outbreak of soybean cyst nematode correlated to drought conditions in north central U.S. (1990)
 - Summer locust outbreak correlated to drought in Mexico (1999)
 - Increased incidence of *Aspergillus flavus* (producer of aflatoxin) in southern U.S. (1977 and 1983)

Air currents

- Air currents provide large-scale transportation for disease agents (e.g., spores of fungi) or insects from overwintering areas to attacking areas
 - The spread of the stem rust that overwinters in Mexico and Texas is favored by moist southern air currents
 - The southern leaf blight of corn spread from Mississippi to the Midwest by air currents of a tropical storm in the Gulf of Mexico during 1970

Warm winters

- Increase overwintering populations of all pests and insect vectors
 - Data reported for the European corn borer, wheat scab, wheat rust, and potato leafhopper
 - Increase population of aphids that carry the soybean mosaic virus
 - Increase population and number of generations of Mexican bean beetle and bean leaf beetle in the U.S.

Source: From Ref.^[3].

CURRENT TRENDS

Global increases in pest-induced losses are observed in all regions and crops since the 1940s.^[1,2] During the same period, there was more than a 33-fold increase in both the amount and toxicity of pesticide use.^[1] The increased pest damage arises from changes in production systems, enhanced resistance of some pests to pesticides, and the production of crops in warmer and more humid climatic regions where crops are more susceptible to pests. The ranges of several important insects, weeds, and pathogens have extended and expanded northwards.^[3] Recent climate trends and extreme weather events may be directly and indirectly contributing to the increased pest damage.^[3,4] Whether the change in global climate has contributed to these observations remains a research question.

There have been several attempts to establish correlations between historic pest damage and climate

conditions.^[3,4] Major pest outbreaks have occurred during favorable weather conditions in the region (Table 1). Records of potato leaf roll in North America from 1930 to 1991 suggest that the outbreaks of this aphid-borne viral disease are related to drought conditions. A 100-year record of the locust behavior in Kansas (1854–1954) shows that the most severe damage was caused during dry years. Climate conditions during El Niño and La Niña years seem to be correlated to pest damage in some regions (e.g., wheat stem rust damage in the U.S. Great Plains; wheat stripe rust epidemics in the U.S. Northwest). Insect damage to soybeans increased during the severe drought of 1988 in the U.S. Midwest. An estimated 3.2 million hectares were sprayed with insecticides to control spotted spider mites across the region and losses to Ohio farmers were estimated to be \$15 to 20 million. (The overall damage of the drought required a \$3-billion bailout by Congress of affected farmers.) Flooding in the summer of

1993 affected 16,000 square miles of farmland in central U.S. Excess wetness increased pathogen outbreaks, particularly in Iowa's cropland in low-lying soils.

FUTURE PROJECTIONS

Human activities are causing the natural atmospheric greenhouse effect to be augmented. The earth's global average temperature over the last century has risen about 0.5°C. Such a warming trend and changes in extremes cannot but affect biophysical processes, the regional incidence of weeds, insects, and crop pathogens, and indeed the entire thermal and hydrological regimes governing our agricultural systems. Although predictions of future climate with models are still tentative and should not be accepted uncritically, they indicate that the anthropogenic forcing will bring about changes in the magnitude and frequency of all key components and natural cycles of the climate system. Most analyses concur that in a changing climate, pests may become even more active than they are currently, expanding their geographical range, and may engender increased use of agricultural chemicals that carry health, ecological and economic costs.^[5,6]

Global climate models predict an overall increase in mean global precipitation and potential for changed hydrological regimes (either drier or wetter) in most places. For crop-pests interactions, a change in the patterns of precipitation may be even more important than an equal change in the annual total. The water regime of pests is also vulnerable to a rise in the daily rate and potential seasonal pattern of evapotranspiration, brought on by warmer temperature, dryer air, or windier conditions. Projected temperature increases can induce earlier and faster development of crops, and cause increased pest damage at the sensitive earlier stages of crop development. Disproportionate warming at high latitudes and high elevations, in winter and nighttime, can all affect crop development, bringing re-patterning of the geographical distribution of

production activities, and alter the ecological balance between the crops and its associated pests.

Even without climate change, pest management faces some serious challenges in the coming decades. The most striking of these are the increasingly high dependence on chemical treatments, and rising costs due to environmental protection and public health policies. Improved climate forecasts can help farmers prepare for changing seasonal-to-interannual conditions, and optimize pesticide management while minimizing environmental damage.

REFERENCES

1. Pimentel, D. Pest management in agriculture. In *Techniques for Reducing Pesticide Use: Environmental and Economic Benefits*; Pimentel, D., Ed.; John Wiley and Sons: Chichester, United Kingdom, 1997; 1–12.
2. Oerke, E.C.; Dehne, H.W.; Schohnbeck, F.; Weber, A. *Crop Production and Crop Protection: Estimated Losses in Major Food and Cash Crops*; Elsevier: Amsterdam, 1994; 830 pp.
3. Rosenzweig, C.; Iglesias, A.; Yang, X.B.; Chivian, E.; Epstein, P. *Implications of Climate Change for U.S. Agriculture: Extreme Weather Events, Plant Diseases, and Pests*; Center for Health and the Global Environment, Harvard Medical School: Cambridge, Massachusetts, 2000; 56 pp.
4. Yang, X.B.; Scherm, H. El Niño and infectious disease. *Science* **1997**, *275*, 739 pp.
5. Intergovernmental Panel on Climate Change (IPCC). In *Climate Change 1995: The Science of Climate Change*; Houghton, J.T., Meira-Filho, L.B., Callander, B.A., Harris, N., Kattenberg, A., Maskell, K., Eds.; Cambridge University Press: Cambridge, United Kingdom, 1996; 572 pp. *Impacts, Adaptations and Mitigation of Climate Change: Scientific-Technical Analyses*; Watson, R.T., Zinyowera, M.C., Moss, R.H., Eds.; Cambridge University Press: Cambridge, United Kingdom, 1996; 879 pp.
6. Coakley, S.M.; Scherm, H.; Chakraborty, S. Climate change and plant disease management. *Annu. Rev. Phytopathol.* **1999**, *37*, 399–426.

Coconut Insects: Ecology and Control

F. W. Howard

Fort Lauderdale Research and Education Center, University of Florida,
Fort Lauderdale, Florida, U.S.A.

INTRODUCTION

The coconut palm, *Cocos nucifera* L. (Palmae) (Fig. 1), grown throughout the lowland tropics, has a prominent place in international agriculture. The fruit of this palm, known as the coconut (Fig. 2), provides raw materials, including: (i) the fibers of the husk, commercially known as coir; (ii) the shell; (iii) the solid white endosperm (kernel); and (iv) the liquid portion of the endosperm known as coconut water. The kernel is an important food source locally, and is marketed internationally. Fresh coconut kernel deteriorates rapidly, but it may be dried to a form known as copra, which can be stored for long periods. Copra is the source of coconut oil, which is among the 10 most important edible oils in world commerce. Minor uses of coconut palm are coconut water as a beverage, fiber products made from the fronds, and “wood” products made from the trunk. The coconut palm is also an important landscaping plant in regions where it is adapted. Coconuts are grown as a crop plant on a total of about 12 million hectares distributed in at least 90 tropical countries. More than 93% of the land area devoted to this crop is in the Asia-Pacific area.^[1]

COCONUT PESTS

Coconut growing requires relatively low input, but in each region there are usually at least several arthropod pests present. Most of the insect pests of coconut are in the orders Orthoptera, Phasmatodea, Hemiptera, Coleoptera, and Lepidoptera. Eriophyidae (Acari) also contains several pests (Table 1).^[2,3]

A few arthropod pests are virtually restricted to coconut palms, e.g., the coconut mite, *A. guerreronis* Keifer (Acari: Eriophyidae), but generally the host ranges of coconut pests include additional species of palms, and sometimes arborescent monocotyledons such as Pandanaceae. Other pests are highly polyphagous. For example, the coconut scale, *A. spidiotus destructor* Signoret (Hemiptera: Diaspididae) has been reported on 75 genera of plants, including monocotyledons and dicotyledons.^[4]

Many coconut pests in Aleyrodidae and Coccoidea, which easily survive on plants transported between

countries and are difficult to detect, have been disseminated widely into several regions. With some important exceptions, most pests of coconut in the orders Orthoptera, Phasmatodea, Coleoptera, and Lepidoptera are not present beyond their native ranges.

Some insect pests of coconut palm, e.g., *A. argaula* (Meyrick) (Lepidoptera: Agonoxenidae), are present only in the South Pacific, the native home of coconut. Others are native to regions where the coconut palm is an exotic. For example, *B. sophorae* L. (Lepidoptera: Nymphalidae) is native to South America, where its original hosts were native palm species. With the introduction of coconut palm and expansion of plantations since the 16th century in that region, it became a pest of coconut.

Although some species of insects and mites attack the coconuts (i.e., the fruits) and thus cause direct damage to the main commercial product, there are far more species that feed on leaves and thus affect the vigor of palms and indirectly the fruit production. These include defoliators and piercing-sucking insects. In coconut palms grown as landscaping plants, conspicuous damage owing to defoliators may be more undesirable than damage to the fruits. Some species, particularly Coccoidea, may occur on both the fronds and the fruits.

Species of eight families of Lepidoptera cause significant damage as externally feeding defoliators of coconut palm. Sixty-four species of Limacodidae were reported on coconut palms in Southeast Asia.^[5] Limacodid pests of coconut palm are also present in Africa and Tropical Asia, but with fewer species.^[6] The other seven families are represented on coconut palms by one to a few species each. Species of the more primitive lepidopterous taxa (Psychidae, Gelechioidea, and Zygaenidae) tend to consume only superficial tissues of the abaxial surface, causing elongate scars on the abaxial leaf surfaces, and necrotic areas opposite them on the adaxial surface. Dense populations of such caterpillars can kill entire fronds (Figs. 3 and 4). Species of more advanced taxa (Limacodidae, Hesperidae, and Nymphalidae) consume portions of the entire lamina. The feeding of dense populations during outbreaks can strip all but the mid-veins of leaflets.

In addition to external defoliators, leaf-mining beetles (Coleoptera: Chrysomelidae: Hispinae) are important pests of coconut palms. In the South Pacific,



Fig. 1 A coconut palm, *C. nucifera* L.

at least 14 species attack coconut palm.^[7] Fewer species of Hispinae are pests of coconut palm in Africa and Tropical America.

Several species each of Tettigoniidae (Orthoptera) and Phasmatodea are defoliators of coconut palm in Oceania.

The insect order with the greatest number of species reported from palm foliage is Hemiptera. The suborder



Fig. 2 A coconut fruit, showing, from exterior toward interior, husk, shell, and kernel.

Heteroptera is poorly represented on coconut palm foliage. Auchenorrhyncha is better represented; species of Fulgoroidea are particularly common on coconut palms. They are rarely numerous enough to cause significant direct damage, but two fulgoroids are reported as vectors of coconut diseases, viz., *M. crudus* Van Duzee and *M. taffini* Bonfils (Cixiidae), vectors of lethal yellowing in Florida and the Caribbean Region and of coconut foliar decay in Vanuatu, respectively.

The great majority of hemipterous species on coconut are species in the suborder Sternorrhyncha, concentrated in Aleyrodidae, and the coccoid families, Pseudococcidae, Coccidae, and Diaspididae. Only two species of aphids (Aphididae) are coconut pests. These are in the genus *Cerataphis*, of the small, specialized subfamily Hormaphidinae.^[8]

Relatively few insects directly damage coconut fruits, probably at least partly owing to the protection provided by the thick, fibrous husk. However, in the tropics of the Eastern Hemisphere, several species of Coreiidae and a species of Pentatomidae (Hemiptera: Heteroptera) puncture young coconuts, resulting in distorted fruit growth and sometimes in premature shedding. Several species of eriophyid mites feed on the meristematic tissue beneath the perianth of coconuts, causing suberization and deformation of the growing coconut. The most damaging of these is *A. guerreronis*, which has long been distributed in the coconut-growing regions of West Africa and the Americas, and in recent years has become established in India and Sri Lanka.^[9–11]

Various species are borers of petioles or other green parts, or the trunks, and sometimes in several of these plant parts. There are relatively few insect species that are primary trunk borers, but these occasionally can be serious pests in some regions. Many palm trunk borers, e.g., the palm weevil, *R. palmarum* F., and the red palm weevil, *R. ferrugineus* Olivier, attack relatively soft, green tissue of the palm such as the petioles or bud, from which the larvae bore into stem tissue. The palm weevil's more serious damage is as a vector of a nematode, *Bursaphelenchus cocophilus* (Cobb), that causes red ring disease of coconut.^[12] Opportunistic borers such as ambrosia beetles sometimes excavate galleries in the trunks of stressed and dying coconut palms. *Sufetula* sp. (Lepidoptera: Pyralidae) is one of only a few significant arthropod pests of coconut roots.^[13]

CONCLUSIONS

Although coconut palms are adapted to marginal lands and can often be grown with little maintenance, they are sometimes severely affected by arthropod pests or by diseases transmitted by them.

Table 1 Examples of insect and mite pests of coconut palm

| Order | Family | Examples | Type of pest | Geographic distribution |
|--------------------------|----------------|---|---|--|
| <i>Insects (Insecta)</i> | | | | |
| Orthoptera | Tettigoniidae | <i>Sexava coreacea</i> L. | Defoliator | Pacific |
| Phasmatodea | | <i>Graeffea crouani</i> Le Guillou | Defoliator | Pacific |
| Hemiptera | Coreidae | <i>Amblypelta cocophaga</i> China | Piercing insect on fruits | Pacific |
| | Pentatomidae | <i>Axiagastus cambelli</i> Distant | Piercing insect on fruits | Pacific |
| | Cixiidae | <i>Myndus crudus</i> Van Duzee | Vector of lethal yellowing | Florida, Caribbean Region |
| | | <i>Myndus taffini</i> Bonfils | Vector of coconut foliar decay | Vanuatu |
| | Aphididae | <i>Cerataphis brasiliensis</i> (Hempel) | Piercing-sucking insect on foliage | Cosmopolitan |
| | Aleyrodidae | <i>Aleurodicus cocois</i> (Curtis) | Piercing-sucking insect on foliage | Southeast Asia |
| | | <i>Aleurodicus destructor</i> Mackie | Piercing-sucking insect on foliage | Tropical America |
| | Pseudococcidae | <i>Nipaeococcus nipae</i> (Maskell) | Piercing-sucking insect on foliage, fruits | Cosmopolitan |
| | Coccidae | <i>Eucalymnatus tessellates</i> (Signoret) | Piercing-sucking insect on foliage | Cosmopolitan |
| | Diaspididae | <i>Aspidiotus destructor</i> Signoret | Piercing-sucking insect on foliage, fruits | Cosmopolitan |
| Coleoptera | Scarabaeidae | <i>Oryctes rhinoceros</i> L. | Borer in bud (adult) | Asia-Pacific |
| | Curculionidae | <i>Rhynchophorus palmarum</i> F. | Borer in bud (larva) | Tropical America |
| | | <i>Rhynchophorus ferrugineus</i> (Olivier) | Borer in bud (larva) | Southeast Asia, Mediterranean |
| | Chrysomelidae | <i>Brontispa longissima</i> Gestro <i>Promecotheca coeruleipennis</i> (Blanchard) | Leaf miner Leaf miner | Pacific Pacific |
| Lepidoptera | Psychidae | <i>Oiketicus kirbyi</i> Guilding <i>Mahasena corbetti</i> Tams | Defoliator Defoliator | Tropical America Asia-Pacific |
| | Coleophoridae | <i>Homaledra</i> sp. | Defoliator | Florida, Caribbean |
| | Oecophoridae | <i>Opisina arenosella</i> Walker | Defoliator | Southern Asia |
| | Agonoxenidae | <i>Agonoxena argaula</i> Meyrick | Defoliator | Pacific |
| | Castniidae | <i>Castnia daedalus</i> Cramer | Stem and petiole borer | South America |
| | Zygaenidae | <i>Artona catoxantha</i> Hampson | Defoliator | Southeast Asia |
| | | <i>Homophylotis catori</i> Jordan | Defoliator | West Africa |
| | Limacodidae | <i>Setora nitens</i> Walker | Defoliator | Southeast Asia |
| | | <i>Parasa viridissima</i> Holland | Defoliator | West Africa |
| | Hesperiidae | <i>Hidari irava</i> Moore & Horsefield | Defoliator | Southeast Asia |
| | Nymphalidae | <i>Amathusia phidippus</i> L. | Defoliator | Southeast Asia |
| | | <i>Brassolis sophorae</i> L. | Defoliator | South America |
| Mites (Acari) | | | | |
| Acariformes | Eriophyidae | <i>Aceria guerreronis</i> Keifer | Gall mite in fruits | Tropical America, West Africa, Southern Asia |

Recently, progress has been made in developing safer methods of chemical control of coconut pests, e.g., a water-soluble neem formulation was shown to protect coconut palms for 120 days from the coconut black-headed caterpillar, *Opisina arenosella* Walker.^[14] A challenge in chemical control research is to develop

methods that are economical in relation to the expected income of coconut plantations.

In coconut breeding, the emphasis has generally been on resistance to diseases, rather than to insects. However, some varieties of coconut palm appear to be resistant or tolerant to certain insects or mites.

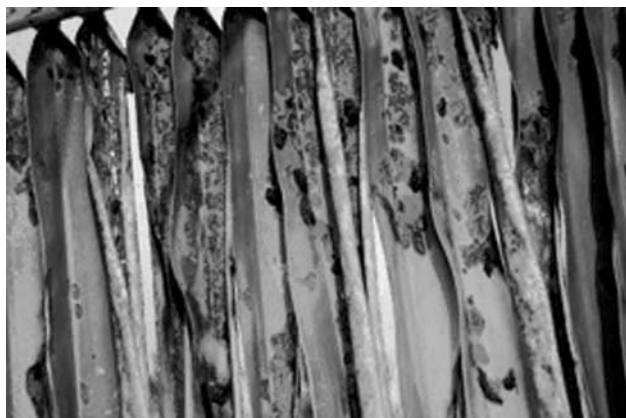


Fig. 3 Damage to a coconut frond by the palm leaf skeletonizer, *H. sabalella*, in Puerto Rico.

For example, the “Kamrupa” variety, recently released in India, is reported to be tolerant to the rhinoceros beetle and the red palm weevil, as well as certain diseases.^[15]

Mass trapping of palm weevils combined with removal of red ring-infected palms effectively reduced the incidence of this disease in African oil palms, and the technique would also apply to coconut palms.^[16] Eliminating dead trunks and other large sources of decaying plant material is a form of cultural control employed to reduce insects that breed in it, e.g., the coconut rhinoceros beetle, *Oryctes rhinoceros* L. (Coleoptera: Scarabaeidae). Maintaining certain plants in the groundcover of coconut plantations may provide nectar sources for natural enemies of coconut pests. Several leguminous plant species that are often used as groundcover in coconut plantations do not support the development of the nymphs of *M. crudus* and their use may reduce populations of this vector of lethal yellowing.^[17]



Fig. 4 Coconut palms with lower fronds desiccated owing to damage by palm leaf skeletonizer.

Biological control has been used to successfully manage many insect pests of coconut palms, especially scale insects, and lepidopterous and coleopterous defoliators. A spectacular example was a biological control campaign in the 1920s that resulted in the virtual elimination of a coconut defoliator, *Levuana iridescens* (Lepidoptera: Zygaenidae) in Fiji.^[18] In other cases, e.g., the coconut black-headed caterpillar, biological control research has progressed for many decades,^[19] but further research and development is needed.

REFERENCES

1. Persley, G.J. *Replanting the Tree of Life*; CAB International: Wallingford, UK, 1992; 156 pp.
2. Howard, F.W.; Moore, D.; Giblin-Davis, R.M.; Abad, R. *Insects on Palms*; CABI Publications: Wallingford, UK, 2001; 400 pp.
3. Howard, F.W.; Giblin-Davis, R.M. Palm insects. In *Encyclopedia of Entomology*; Capinera, J., Ed.; Kluwer Academic Publishers: Dordrecht, The Netherlands, 2004; 1622–1627.
4. Borchsenius, N.S. *A Catalogue of the Armoured Scale Insects (Diaspidoidea) of the World*; Nauka: Moscow, Leningrad, 1966; 449 pp.
5. Cock, M.J.W., Godfray, H.C.J., Holloway, J.D., Eds.; *Slug and Nettle Caterpillars. The Biology, Taxonomy and Control of the Limacodidae of Economic Importance on Palms in South-East Asia*; CAB International: Wallingford, Oxon, UK, 1987; 270.
6. Mariau, D. *Les Limacodidae (Lepidoptera) du palmier à huile et du cocotier. Espèces nuisibles et ennemis naturels*. Plantations, Recherche, Développement **1999**, 6 (3), 149–160.
7. Gressitt, J.L. Hispine beetles of the South Pacific. *Nova Guinea* **1957**, 8 (2), 205–324.
8. Russell, L.M. *Notes on Cerataphis brasiliensis and synonyms palmae, variabilis and fransseni (Homoptera: Aphididae) with a key to Cerataphis species living on palms and orchids*. Proceedings of the Entomological Society of Washington **1996**, 98 (3), 439–449.
9. Moore, D.; Howard, F.W. Coconuts. In *Eriophyid Mites: Their Biology, Natural Enemies and Control*; et al., Ed.; Elsevier Science Publishers B. V: Amsterdam, Lousanne, New York, Oxford, Shannon, Singapore, Tokyo, 1996; 561–570.
10. Sathiamma, B.; Nair, C.P.R.; Koshy, P.K. Outbreak of nut infesting eriophyid mite *Eriophyes guerreronis* (K.) in coconut plantations in India. *Indian Coconut J.* **1998**, 29 (2), 1–3.
11. Varadarajan, M.V.; David, P.M.M. Population dynamics of the coconut mite, *Aceria guerreronis* Keifer (Acari: Eriophyidae) and associated arthropods in Tamil Nadu, India. *Insect Sci. Its Appl.* **2002**, 22 (1), 47–59.
12. Giblin-Davis, R.M. Interactions of nematodes with insects. In *Nematode Interactions*; Khan, M.W., Ed.; Chapman & Hall: London, 1993; 302–344.

13. Bonneau, X.; Husni, M.; Philippe, R.; Somchit, N.; Jourdan, C.; Lubis, N. Discovery of a factor limiting yields in a coconut plantation on peat: the insect pests *Sufetula* sp. *Exp. Agric.* **2004**, *40*, 53–64.
14. Shivashankar, T.; Annadurai, R.S.; Srinivas, M.; Preethi, G.; Sharada, T.B.; Paramashivapa, R.; Rao, A.S.; Prabhu, K.S.; Ramadoss, C.S.; Veeresh, G.K.; Rao, P.V.S. Control of black-headed caterpillar (*Opisina arenosella* Walker) by systemic application of 'Soluneem'—a new water-soluble neem insecticide formulation. *Curr. Sci.* **2000**, *78* (2), 176–179.
15. Chowdhury, D.; Nath, J.C.; Mohan, N.K. 'Kamrupa'—a newly released coconut variety by Assam Agricultural University. *Indian Coconut J.* **2001**, *31* (9), 12–13.
16. Oehlschlager, A.C.; Chinchilla, C.; Castillo, G.; Gonzalez, L. Control of red ring disease by mass trapping of *Rhynchophorus palmarum* (Coleoptera: Curculionidae). *Fla. Entomol.* **2002**, *85* (3), 507–513.
17. Howard, F.W. Evaluation of dicotylenonous herba-ceous plants as hosts of *Myndus crudus* (Hemiptera: Auchenorrhyncha: Cixiidae). *Plantations, Recherche, Développement* **1999**, *6* (2), 95–99.
18. Tothill, J.D.; Taylor, T.H.C.; Paine, R.W. *The Coconut Moth in Fiji*; Imperial Bureau of Entomology: London, 1930.
19. Desai, V.S.; Narangalkar, A.L.; Nagwekar, D.D. Biological control of coconut blackheaded caterpillar, *Opisina arenosella* Wlk. *Indian Coconut J.* **2003**, *34* (7), 6–8.

Coffee Insects: Ecology and Control

Fernando E. Vega
Francisco Posada

*Insect Biocontrol Laboratory, United States Department of Agriculture,
Agricultural Research Service, Beltsville, Maryland, U.S.A.*

Francisco Infante

El Colegio de la Frontera Sur (ECOSUR), Tapachula, Chiapas, Mexico

INTRODUCTION

Endemic to Africa and now grown in more than 11 million hectares in over 70 countries in the tropics, coffee is the most important agricultural commodity in the world, with an annual estimated retail value of over \$70 billion. Approximately 17–20 million families throughout the world depend on coffee for their subsistence, and total production per year is around 115 million 60-kg bags. The genus *Coffea* consists of over 90 species^[1] but only two species, *Coffea arabica* L. and *Coffea canephora* Pierre ex Froehner (also known as robusta), are commercially traded, with *C. arabica* comprising approximately 65% of coffee production.^[2] Several *C. arabica* cultivars are grown (e.g., Typica, Bourbon, Catuai, Caturra, Maragogipe, Mundo Novo), but their genetic base is small because of a narrow gene pool from which they originated and the fact that they are self-pollinated (i.e., self-fertile) in contrast to *C. canephora*, which is cross-pollinated (i.e., self-sterile). *C. arabica* tends to do better at higher elevations, while robusta is more suited to lower elevations. Nevertheless, both are susceptible to fungal and insect pests. More than 850 insects have been reported to attack coffee.^[3,4] Of these, the most important significant throughout the world are the coffee leaf miner, the coffee berry borer, and the coffee stem borers.

THE COFFEE LEAF MINER [*LEUCOPTERA* *COFFEELLA* (GUÉRIN-MÉNEVILLE) (LEPIDOPTERA: LYONETIIDAE)]

Two different genera are used in the scientific literature dealing with the coffee leaf miner: *Leucoptera* and *Perileucoptera*. To address this unusual situation, we present a short summary on the systematics of this insect. The coffee leaf miner was first described as *Elachista coffeella* by Guérin-Méneville^[5] using specimens collected in Martinique and Guadeloupe. Stainton^[6] placed it in the genus *Bucculatrix* and later on

classified it as *Cemiostoma coffeella*,^[7] while Mann^[8] referred to this species as *Cemiostoma coffeellum*. Walsingham^[9] placed it in the genus *Leucoptera* and Silvestri^[10] transferred it to the genus *Perileucoptera*. Bradley^[11] remarked on differences in wing venation between the specimens used by Silvestri and the specimens he used from Trinidad and concluded that because of these differences “this genus has not been adopted” (by him) and used *L. coffeella*. Even though some papers state that Mey’s^[12] monograph recognizes *P. coffeella* as a junior synonym of *L. coffeella*, this is not an accurate statement (W. Mey, personal communication); therefore, both *Perileucoptera* and *Leucoptera* are valid until a formal phylogenetic analysis and formal synonymization is published. For many years it was thought that *L. coffeella* occurred in East Africa, but after comparing specimens from Trinidad and East Africa, Bradley^[11] concluded that the East African species had been erroneously identified and should be classified as *L. meyricki* Ghesquière. Two other *Leucoptera* species are known in Africa: *L. caffena* and *L. coma*. The area of origin of *L. coffeella* is unknown, although it has been hypothesized that it entered the American continent through plants brought from the island of Réunion.

The coffee leaf miner, a micromoth measuring approximately 2 mm in length, is the most important pest of coffee in Brazil, and is widely distributed throughout the American continent. Eggs are laid on the adaxial side of leaves, followed by larvae mining into the leaves and consuming the mesophyll eventually creating necrotic lesions that reduce photosynthetic area and consequently yields. Losses of up to 50% have been reported in Brazil and 40% in Puerto Rico. The insect does better in dry conditions and high temperatures and can have 7–12 generations per year. Plants in wet areas have very low infestation levels because of water entering the mine and drowning the larvae. Before pupating, the larva emerges from the mine, spins a thread, and using the wind balloons to other plants, where it spins a cocoon usually on the abaxial side of the leaf. The insect can be mass reared in vitro using detached coffee leaves.

Reliance on insecticides has had a detrimental effect on natural enemies and has resulted in the development of resistance to various organophosphates (e.g., disulfoton, ehtion, methyl-parathion, chlorpyrifos). The coffee leaf miner has at least 10 predatory wasps (Vespidae), 21 larval parasitoids (Eulophidae), and eight larval-pupal parasitoids (Braconidae). The insect is susceptible to various endotoxins produced by *Bacillus thuringiensis*, and transgenic *C. arabica* and *C. canephora* plants expressing the *B. thuringiensis* cry1Ac have been developed by French scientists. These were planted in French Guiana in 2000 and were cut down by vandals in 2005 although preliminary data indicated that 70% of the transgenic trees were completely resistant to the insect. Traditional breeding methods are being pursued in Brazil to develop varieties resistant to the coffee leaf miner.

The coffee leaf miner sex pheromones (5,9-dimethylpentadecane and 5,9-dimethylhexadecane) have been identified, and their use in the field has been proposed as a male-confusion technique. Field studies have shown that most captures in pheromone traps occur at midday. The proper management of shade and fertilization, minimizing the use of insecticides, and the conservation of natural enemies are important factors to reduce coffee leaf miner outbreaks in coffee plantations.

THE COFFEE BERRY BORER [*HYPOTHENEMUS HAMPEI* (FERRARI)] COLEOPTERA: CURCULIONIDAE]

The coffee berry borer, a coffee specialist, is endemic to Central Africa and has now been reported in most coffee producing countries, with the notable exceptions of Hawaii and Puerto Rico. A phylogenetic analysis by Benavides et al.^[13] using specimens from 17 countries revealed that only one species is present. Females bore a hole in the coffee berry and deposit their eggs inside; larval feeding on the endosperm greatly reduces quality and yields and can also cause abscission of the berry. Sibling mating occurs inside the berry, and 10 females are produced for every male, most likely because of the presence of the bacterium *Wolbachia*. Once adult females have emerged from the berry they are inseminated and immediately attempt to locate another berry in which to oviposit. This makes the use of insecticides very ineffective because of the short window of time during which the insect is outside the berry. Several insecticides have been used, including endosulfan, to which the insect has developed resistance. The effects of shade on the coffee berry borer are equivocal: da Fonseca^[14] reported increased incidence in coffee grown under heavy shade while Soto-Pinto, Perfecto, and Caballero-Nieto^[15] reported

no correlation between shade/light and infestation levels. Overall, the effects of shade on insect pests and plant diseases are very complex because of their different environmental requirements for successful colonization and reproduction.^[16] Classical biological control research programs have been conducted in several countries against the coffee berry borer, and parasitoids from Africa [e.g., *Prorops nasuta* Waterston, *Cephalonomia stephanoderis* Betrem (Hymenoptera: Bethyloidea), and *Phymastichus coffea* LaSalle (Hymenoptera: Eulophidae)] have been introduced to other coffee growing regions. Various fungal entomopathogens, such as *Beauveria bassiana* Balsamo (Vuillemin), *Metarhizium anisopliae* (Metschnikoff) Sorokin, *Hirsutiella eleutheratorum* (Nees) Petch, *Paecilomyces fumosoroseus* (Wize) Brown & Smith, and *P. lilacinus* (Thom) Samson, have been isolated from the insect. Growers in many countries grow *B. bassiana* and spray it in their plantations. Two nematodes have been reported attacking the insect: *Panagrolaimus* sp. in India, and *Metaparasitylenchus hypothenemi* Poinar et al. in Mexico.

COFFEE STEM BORERS

Several cerambycids are considered serious pests of coffee, because of larval stages boring into the trunk. These are discussed below:

Monochamus leuconotus (Pascoe)

Known as the white coffee stem borer, this insect has been a pest of coffee in eastern, central, and southern Africa for over 100 years. Eggs are laid on the trunk, and young larvae ringbark the trunk and roots, frequently causing death of the tree. Older larvae bore into the stem and feed for several months. Adult beetles, which are not attracted to light, feed on newly flushed leaf tissue but do not cause major damage to these. Eulophids, braconids, pteromalids, scelionids, and other parasitic Hymenoptera have been reported as natural enemies of this insect.

Bixadus sierricola (White)

It is an important pest in Central and West Africa. Eggs are laid on the bark; young larvae ring the bark and older larvae bore into the trunk where they feed for several months, producing large amounts of wood shavings and frass, which fall at the base of the tree under the entrance hole. Young trees usually die because of the ringbark damage, and older plants can topple over with the wind or become susceptible to termites and fungi. Adults, which feed on the bark of

green shoots, are poor fliers and are strongly attracted to light. An ichneumonid and a tachinid are known to parasitize larvae of this insect.

***Xylotrechus quadripes* (Chevrolat)**

A serious pest of coffee in South-east Asia and India. Eggs are laid on the bark, and larvae entering the bark make tunnels, which create ridges on the bark surface that are used as an indication of infestation. Adults are strong fliers, and several parasitoids have been reported attacking this insect, including bethylids, braconids, eurytomids, evaniids, and ichneumonids. Birds have also been reported as predators of larval stages, and low infestations (~2.5%) with the fungal entomopathogen *B. bassiana* have been reported in India.

***Plagiohammus* sp. and *Neoclytus cacticus* (Chevrolat)**

Plagiohammus sp. has been reported attacking coffee trees in Mexico, Guatemala, El Salvador, Honduras, and Costa Rica. The life cycle is about 20 months, and adults typically emerge from the stems between April and June each year. Larval feeding can delay plant growth and development and in extreme cases kills the plant or makes it susceptible to falling down. *N. cacticus* has been reported attacking coffee plants in Guatemala.

COFFEE STEM BORER MANAGEMENT TECHNIQUES

Insecticides have been used in an attempt to control these insects, but because of their cryptic life cycle inside the trunk, the effectiveness of such method is doubtful. A paint containing an insecticide, which can be applied on the stem to kill the eggs and larvae as they bore, has been suggested as a possible method for control. For example, *M. leuconotus* was successfully controlled in the 1950s with 2% dieldrin paint applied to the base of the stems, but needless to say, use of methods such as this, based on highly toxic poisons, presents problems to both humans and the environment. For *B. sierricola*, fumigants have been inserted into the bores made by the insects as a control tactic. Among these, a paste containing aluminum phosphide was placed in the holes of 3200 trees attacked by stem borers in Ghana and sealed with plasticine, resulting in 100% mortality. This method relies on a highly dangerous chemical that has to be applied by hand in trees that have already been attacked. Cultural practices have been used, but these require

intensive labor, e.g., collecting and killing adult insects, manually killing larvae with a wire inserted in the hole, and uprooting and burning of infested trees.

CONCLUSIONS

Owing to the low coffee prices that were prevalent in the market for several years, small coffee growers were for the most part not able to invest in pest management strategies that required inputs external to the farm, i.e., insecticides. This, on the one hand, resulted in the production of coffee that could be considered organic—even though the term “organic” implies more than not using pesticides—but on the other hand, led to many growers having to abandon coffee harvesting because of the severe losses caused by insect pests. The prospects, in terms of implementing innovative pest management strategies that are inexpensive and sustainable, remain bleak in great part because of scarce research funds in coffee-producing countries and the lack of an organized structure that oversees coffee research throughout the world. Research aimed at developing innovative biological control methods against coffee insects should be promoted and encouraged by major coffee companies that, after all, have a tremendous stake and interest in high quality coffee. Successful biological control of insect pests in coffee plantations could result in reduced expenses for small coffee growers who cannot fund or do this research on their own. One particularly innovative area of research involves establishing fungal entomopathogens as coffee endophytes; if successful in controlling insects, it would be a revolutionary pest management strategy.

REFERENCES

1. Bridson, D.M.; Vercourt, D. *Coffea* and *Psilanthus*. In *Flora of Tropical East Africa: Rubiaceae, Part 2*; Poolhill, R.M., Ed.; Balkema: Rotterdam, 1988; 703–727.
2. Lewin, B.; Giovannucci, D.; Varangis, P. *Coffee Markets: New Paradigms in Global Supply and Demand*, The International Bank for Reconstruction and Development, Agriculture and Rural Development Discussion Paper 3; The World Bank: Washington, DC, 2004, 149.
3. Le Pelley, R.H. Coffee insects. *Annu. Rev. Entomol.* **1973**, *18*, 121–142.
4. Le Pelley, R.H. *Pests of Coffee*; Longmans, Green and Co., Ltd.: London, 1968; 590 pp.
5. Guérin-Méneville, F.E. *Memoire sur un Insecte et un Champignon que ravagent les Caféiers aux Antilles*; Ministère de la Marine: Paris, 1842; 40 pp.

6. Stainton, H.T. Suggestions respecting *Tineina* for a traveler in South America. *Entomol. Weekly Intell.* **1858**, 4, 54–70.
7. Stainton, H.T. A few words respecting *Cemiostoma coffeella*; an insect injurious to coffee plantations of the West Indies. *Entomol. Weekly Intell.* **1861**, 10, 110–111.
8. Mann, B.P. The white coffee-leaf miner. *Am. Nat.* **1872**, 6, 596–607.
9. Walsingham, Lord. Revision of the West-Indian Microlepidoptera. In *Proc. Gen. Mtngs. for Sci. Bus., Zool. Soc.*; London, 1897; 54–182.
10. Silvestri, F. *Compendio di entomologia applicata*; Parte speciali; Portici, Italy, 1943; Vol. II (Fogli 1-32), 512.
11. Bradley, J.D. Taxonomic notes on *Leucoptera meyricki* Ghesquiere and *Leucoptera coffeella* (Guérin-Méneville) (Lepidoptera, Lyonetiidae). *Bull. Ent. Res.* **1958**, 49, 417–419.
12. Mey, W. Taxonomische Bearbeitung der westpaläarktischen Arten der Gattung *Leucoptera* Hübner, [1825], s.l. (Insecta, Lepidoptera). *Deut. Entomol. Zeit. N.F.* **1994**, 41, 173–234.
13. Benavides, P.; Vega, F.E.; Romero-Severson, J.; Bustillo, A.E.; Stuart, J. Biodiversity and biogeography of an important inbred pest of coffee, the coffee berry borer, *Hypothenemus hampei* (Ferrari). *Ann. Entomol. Soc. Am.* **2005**, 98, 359–366.
14. da Fonseca, J.P. A 'broca' e o sombreamento dos cafezias. *Biológico* **1939**, 5, 133–136.
15. Soto-Pinto, L.; Perfecto, I.; Caballero-Nieto, J. Shade over coffee: its effects on berry borer, leaf rust and spontaneous herbs in Chiapas, Mexico. *Agroforest. Syst.* **2002**, 55, 37–45.
16. Beer, J.; Muschler, R.; Kass, D.; Somarriba, E. Shade management in coffee and cacao plantations. *Agroforest. Syst.* **1998**, 38, 139–164.

Colorado Potato Beetle: Thermal Control

Mohamed Khelifi

Department of Soil Science and Agri-Food Engineering, Université Laval, Sainte-Foy, Quebec, Canada

Raymond-Marie Duchesne

Direction de l'Environnement et du Développement Durable, Quebec Ministry of Agriculture, Fisheries, and Food, Sainte-Foy, Quebec, Canada

Claude Laguë

College of Engineering, University of Saskatchewan, Saskatoon, Saskatchewan, Canada

Jacques Gill

Department of Soil Science and Agri-Food Engineering, College of Agriculture and Food Sciences, University of Laval, Quebec, Quebec, Canada

INTRODUCTION

The use of heat to control Colorado potato beetle (CPB) is a relatively recent technical innovation. Its efficacy at controlling spring CPB adults was first demonstrated in 1989 in the United States where a mortality rate of 80% was obtained. Between 1991 and 1995, many research and development studies on that topic were conducted in North America, mainly in the State of New York, U.S.A. and in Quebec, Canada.

Thermal control of CPB constitutes an alternative to the use of chemical insecticides and can contribute to the reduction of the problems associated to the contamination of underground and surface water by pesticide residues. In addition, it does not have any residual effect on the crop. The use of this technique also reduces the risk of developing insecticide-resistant populations of CPB.

IMPACT OF HEAT ON COLORADO POTATO BEETLE

The use of heat to control CPB can result in a rapid death following a violent thermal shock or in sublethal injuries that affect the insect behavior. The first symptom of a thermal shock on the insect is the coagulation of the membrane proteins when the temperature of the cellular tissues reaches the 50–70°C range. Temperatures of about 70°C can generate sufficient muscle injuries to reduce the insect mobility and consequently its ability to feed and to reproduce, ultimately leading to death. Body segments having the smallest diameters are first damaged; tarsi, antennae, femur–tibia joints, and coxa–body joints are thus sequentially affected. In addition, the application of heat will lead to partial or total destruction of the exposed egg masses.

Efficacy of Thermal Control of Colorado Potato Beetle

In 1994, an important research program related to the thermal control of CPB was initiated in Quebec, Canada, by a multidisciplinary team. Trials were conducted in the laboratory using dedicated facilities at the Université Laval and in the field, both in experimental plots and under commercial conditions. Thermal treatments were applied using appropriately designed propane flamers.

Laboratory tests showed a good tolerance of the young potato plants to thermal treatments that resulted into exposition temperatures of 175°C, which correspond to the level required for an efficient control of adult CPB. Using plant height and the presence of external damage as indicators, it was found that younger plants, less than 10 cm tall, better resisted to heat and recovered more rapidly. These tests also showed that CPB eggs were more sensitive to heat than adult insects. For temperatures in the 75–200°C range, all CPB larval stages were highly sensitive to heat with a mortality rate of 100%. For CPB adults, exposition to temperatures greater than 100°C resulted in a mortality rate of 57% and more. At temperatures exceeding 150°C, more than 75% of the adults died within 2 days, whereas surviving individuals showed highly reduced mobility and feeding capacity.

Similar results were obtained in the field. Twenty days after the thermal treatments, the differences in heights observed between plants thermally treated and those untreated were 3.2, 6.7, and 9.3 cm in average for the treatments at 175°C applied at the stages of 0 to 5, 5 to 10, and 10 to 15 cm, respectively. Visual index damage of the effects of heat on plants, ranging from 0 when no damage was observed to 10 when

91–100% of the plants were affected, followed the same trend. Ten days after thermal treatments, values of this index were 1.5, 1.5, and 3.5 for plants having heights in the ranges 0 to 5, 5 to 10, and 10 to 15 cm, respectively. Twenty days after applying the thermal treatments, all traces of damage disappeared and all index values were null for the three plant height ranges and all the thermal treatments used.

Field results also showed that the mortality rate of spring adults ranged from 77% to 90%, and that these rates were higher for CPB located on young plants (0 to 5 cm) where they were more exposed to the heat. At a temperature of about 175°C at the plant level, the mortality rate reached up to 80% for adult CPB and 100% for larvae and eggs. In the latter case, eggs that were slightly protected by leaves showed a mortality rate of 5% to 20% lower than those that were directly exposed to heat. This phenomenon of foliage shielding was also observed for other CPB stages and tended to increase with the density of the crop canopy.

Under commercial production conditions, trials conducted on potato plants less than 10 cm tall and targeted at the simultaneous thermal control of emerging CPB and young weeds confirmed that the recovery of plants was comparable to that observed during the previous laboratory and experimental plot tests. In addition, the yield measurements at the end of the season did confirm that such spring treatments, when applied to the young potato plants, did not have any negative impact on yields.

Control of Colorado Potato Beetle During Crop Top Killing

Laboratory thermal treatments applied on the three varieties of mature potato plants (Chieftain, Kennebec, and Superior) grown in greenhouse resulted in plant defoliation similar to that obtained chemically.

Although the measurements of exposition temperature inside the plant foliage did present a large variability, the average temperature of exposition associated to each treatment followed the general equation for treatment intensity (intensity is proportional to fuel pressure in the flammers and inversely proportional to flamer travel speed). Temperatures of exposition required for top killing potato plants were also found to correspond to the heat intensities required for obtaining a mortality rate of 75% for adult CPB (150–200°C). It is therefore possible to reduce the density of CPB population at the end of the season when using the thermal method to defoliate potato plants.

Results obtained from grower's fields in 1995 showed CPB mortality rates of 87% and 92% for the cultivars Snowden and Atlantic, respectively. The quality of the thermal top killing was similar and even better than the conventional chemical defoliation (with REGLONE^{MC}) in all sites, even for varieties as turgescient as Kennebec. For the cultivars Atlantic, Snowden, and Niska that were grown for the chips market, sugar rates and chips coloration tests all indicated an excellent quality of tubers coming from the plots thermally defoliated. Finally, the emergence rate of Kennebec tubers that were being stored during the winter season was not affected.

COMBINED STRATEGY TO CONTROL COLORADO POTATO BEETLE POPULATIONS DURING THE COMPLETE GROWING SEASON

Thermal treatments can be used to control CPB at two specific moments during the growing season of the potato crops: 1) shortly after the potato plants have emerged at a period where the potato plants are more resistant to heat than CPB; and 2) at top killing (Fig. 1). Thermal control has therefore a limited reach and should be complemented with other means of CPB control.

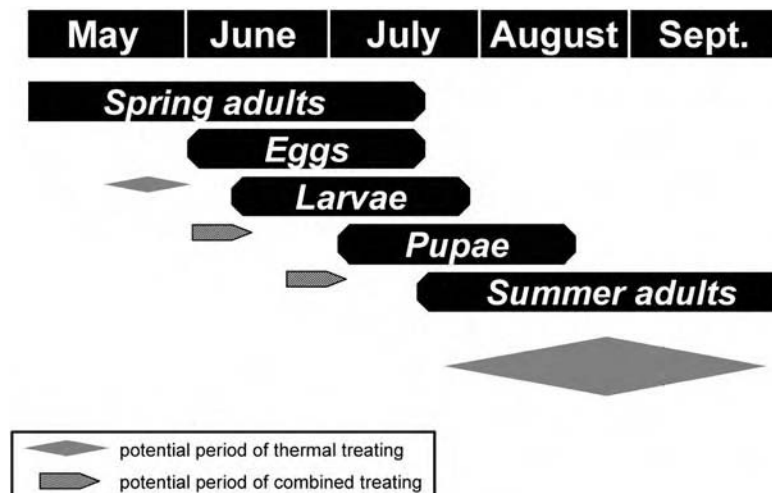


Fig. 1 Potential usage periods of the thermal and combined (thermal and pneumatic) control methods within the seasonal biological cycle of CPB.

One such alternative is to combine the pneumatic and thermal control on a dedicated implement that could be used when the potato plants are more susceptible to heat. Air is blown through the plants to dislodge the insects from the foliage, and the dislodged insects are directed to the ground surface between the crop rows. Shielded flaming units can then be used to destroy the insects without negatively affecting the potato plants.

This combined control strategy was tested on three varieties of potato (Yukon Gold, Superior, and Chieftain) on two commercial farms located in Quebec, Canada, using a four-row prototype specially designed for this purpose. Results showed that the use of this combined implement had no negative effects on the growth of the potato plants. In addition, an improved control of CPB larvae populations was observed. The use of this strategy to control CPB adults was as efficient as the use of chemical insecticides.

EQUIPMENT USE AND COSTS

Spring thermal treatments against CPB must be completed before the potato plants reach a height of 10 cm. It was also observed that irreversible negative effects on plants and decreases in yield occurred if more than one thermal treatment was applied. During top killing, more than one thermal treatment could be applied with no negative effects on the tuber quality.

Total operational costs for spring thermal treatments that could control both the emerging CPB adults and young weeds were evaluated in the range \$52.70 to \$70.90 CDN/ha. Such costs compare favorably with the conventional chemical control methods. For top killing potato cultivars that have a high foliage density, the use of thermal control (operational costs in the \$49.30 to \$105.50 CDN/ha range) is, however, more expensive than chemical defoliants. For the combined pneumatic–thermal implement, total operational costs were estimated at \$25 to \$40 CDN/ha.

FURTHER DEVELOPMENTS

Equipment performances and field capacities remain the major weaknesses of the thermal method. For example, travel speeds of 6 km hr⁻¹ for spring treatments, 5 km hr⁻¹ for combined pneumatic–thermal control later in the season, and 3.5 km hr⁻¹ in average for the thermal top killing operations are, in general, slower than those of the conventional sprayers. In addition, these equipments have a reduced operational width (less than 6 m) which greatly reduces their field capacity compared with sprayers. Costs and energy use for the thermal control systems could be

reduced by making use of systems capable of detecting the insects present on the plants or the weeds on the ground and of controlling the components of the machine.

CONCLUSION

Laboratory and field tests showed that young plants potato plants having a height of 10 cm or less are more resistant to thermal treatments aimed at controlling CPB and recover more rapidly. Thermal control of CPB adults, larvae, and eggs was efficient, in particular for exposition temperatures of about 175°C at the plant level. Thermal control of CPB applied to young potato plants did not have negative impacts on crop yield. The use of thermal treatments at the end of the growing season for top killing of potato plants yielded similar levels of plant defoliation to those achieved with chemical defoliants. In addition, such treatments were very effective at reducing the population of CPB adult insects. Using an integrated pneumatic–thermal method to control the populations of CPB adults during the growing season was found to be as effective as the use of chemical insecticides and thus constitutes an interesting alternative for the control of CPB in potato production.

BIBLIOGRAPHY

- Duchesne, R.-M., Boiteau, G., Eds.; *Potato Insect Pest Control: Development of a Sustainable Approach*; Direction de la recherche, Quebec Ministry of Agriculture, Fisheries, and Food: Quebec, Qc, 1995; 204 pp.
- Gill, J.; Laguë, C.; Lehoux, N.; Péloquin, G. *Test Bench for Thermal Weed and Pest Insect Control*; ASAE Paper 94-8512; American Society of Agricultural Engineers: St-Joseph, MI, USA, 1994.
- Laguë, C.; Bernier, D.; Duchesne, R.-M. *Use of Propane Flamers for Weeds and Colorado Potato Beetle Control and for Top Killing in Potato Crops*; Centre de recherche en horticulture, Université Laval: Quebec, Canada, 1994.
- Laguë, C.; Gill, J.; Lehoux, N.; Péloquin, G. Engineering performances of propane flamers used for weed, pest insect and plant disease control. *Appl. Eng. Agric.* **1997**, *13* (1), 7–16.
- Laguë, C.; Khelifi, M.; Gill, J.; Lacasse, B. Pneumatic and thermal control of Colorado potato beetle. *Can. Agric. Eng.* **1999**, *1* (41), 53–57.
- Moyer, D.D.; Derksen, R.C.; McLeod, M.J. Development of a propane flamer for Colorado potato beetle. *Am. Potato J.* **1992**, *69*, 599–600.
- Pelletier, Y.; McLeod, C.D.; Bernard, G. Description of sublethal injuries caused to the Colorado potato beetle (*Coleoptera: Chrysomelidae*) by propane flamer treatment. *J. Econ. Entomol.* **1995**, *88*, 1203–1205.

Consumer Concerns about Pesticides and Pests

George Ekström

Former Swedish Pesticide Regulator, Solna, Sweden

Margareta Palmberg

Swedish Poisons Information Centre, Stockholm, Sweden

INTRODUCTION

A recent European survey of risk perception and food safety showed, in line with previous research findings, that consumers tend to worry most about risks caused by external factors over which they have little or no control. Consequently, consumers appear to be less worried about risks possibly associated with their own behavior or practices. Physicians and scientists are the most trusted information sources with regard to serious food risks, followed by public authorities and mass media. Economic operators (food manufacturers, farmers, and retailers) are cited as being among the least trusted.^[1]

Interviews conducted with over 1000 consumers in a survey done in 2001 by the British Co-op Group showed that consumers were concerned about the effects of pesticides. Consumers who took part in this survey, when prompted with a series of questions, were concerned that pesticides are harmful to wildlife, leave residues in food, pollute water courses, are harmful to growing children, are harmful to the respondents themselves, and damage the health of farm workers.^[2,3] According to a personal communication with David Pimentel, in the United States, the Food and Drug Administration has reported that 97% of people prefer foods without pesticides.

Particular causes of consumer concern are the potential for ‘cocktail’ effects from multiple residues (see Table 1),^[4-6] and the fact that children may exceed health-related acute reference doses even at legally acceptable residue levels (see Table 2).^[6] In Australia, the Food Standards code contains provisions for an additional, overall limit for pesticides belonging to the same chemical group (see Table 3).^[7]

PESTS AND PESTICIDE SAFETY IN HOMES AND GARDENS

Consumers use a range of pesticides in their homes and gardens:

- *Herbicides* against weeds in vegetables, moss in turf, brush, etc.
- *Fungicides* against mold, mildew, etc.

- *Insecticides* against aphids, greenflies, ants, wasps, pests on potted plants, moths, pantry pests, cockroaches, flies, etc.
- *Rodenticides* against moles, rats, mice, voles, etc.
- *Repellents* against mosquitoes, black flies, ticks, game, and pests on dogs, cats, and horses.
- *Wood preservatives* against rot on timber or furniture.

For the general public, ingestion is the most common route of pesticide exposure. Accidental, single, high-level exposures can lead to acute pesticide poisoning, often in children, and may result from mistakenly swallowed pesticides stored in unlocked cabinets or in unmarked bottles or containers. With regard to long-term, low-level exposure of the general public, the main route of exposure, ingestion through food and drinking water, is followed by inhalation through air or dust. This exposure results in an unknown number of people with diverse chronic health effects.^[8]

A comparison of pesticide poisoning cases in 1984, 1994, and 2004, performed by the Swedish Poisons Information Centre, showed that there was an increase in the overall number of human cases related to pesticide exposure—from 493 in 1984, and 774 in 1994, to 1071 in 2004. The proportion of pesticide-related inquiries, however, remained constant at 3%.

Most incidents were due to accidental exposure at home. Ingestion was the most frequent route of exposure, followed by inhalation. Data from Swedish hospitals reported to the Poisons Information Centre showed the same pattern, that is, an increase in the proportion of cases related to accidental exposure at home. Children were involved in about 60% of all cases in 1984 as well as in 1994 and 2004. With exception of ‘superwarfarins’ found in some rodenticides, most pesticides involved in incidents at home were of low toxicity and present at low concentrations in the formulated products. The few severe cases are mainly intentional poisonings.^[9]

Thirty percent of the total number of inquiries to the Centre in 2004 was due to childrens’ ingestion of insecticides intended for control of ants, containing low concentrations of borax, organophosphorus compounds or pyrethroids. No symptoms were recorded.

Table 1 Multiple residues found in a single sample of pears 2004

| Pesticides found | Residue level (mg/kg) | Maximum residue limit (mg/kg) | Residue level in % of maximum residue limit |
|-------------------------|-----------------------|-------------------------------|---|
| Dithiocarbamates | 0.305 | 3 | 10 |
| Chlorpropham | 0.155 | 0.05 | 310 |
| Azinphosmethyl | 0.084 | 0.5 | 17 |
| Procymidone | 0.071 | 1 | 7 |
| Dichlofluanid | 0.060 | 5 | 1 |
| Chlorpyriphos | 0.059 | 0.5 | 12 |
| Bromopropylate | 0.055 | 0.05 | 110 |
| Cyprodinil | 0.022 | — | — |
| Combined total residues | | | 466 |

Source: Swedish food residue monitoring report. (From Ref.^[6].)

Therefore, the considerable number of inquiries may reflect anxiety about pesticide exposure among the Swedish population.

FOOD RESIDUES

Pesticide Residues in Foods from Organic, Integrated and Conventional Production^[6]

In the Swedish monitoring program for 2004, no residues were detectable in 57% of the samples. Residues at or below maximum residue limits (MRLs) were found in 39% of the samples. 3.5% of all samples contained residues above the MRLs. Of foods from organic production, 4%–5% (import and domestic, respectively) contained detectable residues. Foods from integrated production were free from detectable residues in 91% of domestic produce and 50% of imported product. Foods from conventional production contained no detectable residues in 83% of domestic foods and 46% of imported foods. Residues below the MRLs were found in foods from all three production categories. Residues above the MRLs were found only in imported products from conventional production. No residues were found in any of the 92 samples of foods intended for infants and young children.

Although produced without pesticides, organically produced foods sometimes contain residues. The reason for this may be unintentional mix-up of foods from different sources (organic and conventional), environmental contamination of soils and plants, or fraud. The organic foods that contained pesticides in the United States have been shown to come mostly from soils treated many years ago with DDT or arsenical compounds, according to a personal communication with David Pimentel.

In 11 food commodities (22 samples) from ten countries, residues of ten different pesticides were found at levels 10–37 times the MRL. Multiple residues were found in 492 samples of which 279 samples with two residues, 127 samples with three residues, 54 samples with four residues, 25 samples with five residues, five samples with six residues, and two samples with eight residues (see Table 1) (Fig. 1).

TOWARDS RESIDUE-REDUCED FOOD CROPS

Government Action Plans

The British Food Standards Agency has recognized that while levels of pesticide residues typically found

Table 2 Food residues potentially leading to short time intake in excess of the acute reference dose (ARfD) for toddlers 2004

| Pesticide | Food commodity | Highest residue found (mg/kg) | Maximum residue limit (mg/kg) | ARfD (mg/kg body weight) | Intake, % of ARfD for toddlers |
|--------------------|------------------|-------------------------------|-------------------------------|--------------------------|--------------------------------|
| Dicofthophos | Chinese broccoli | 4.14 | — | 0.0017 | 1763 |
| Lambda-cyhalothrin | Lettuce | 0.92 | 1 | 0.0075 | 106 |
| Oxamyl | Cucumber | 0.42 | — | 0.009 | 135 |
| Endosulfan | Melon | 0.21 | 0.3 | 0.02 | 110 |
| Monocrothophos | Zucchini | 0.14 | — | 0.002 | 381 |
| Aldicarb | Potatoes | 0.035 | 0.5 | 0.003 | 122 |

Source: Swedish food residue monitoring report. (From Ref.^[6].)

Table 3 Approaches to the limitation of organophosphorus pesticide residues in food in Australia and by the British Co-operative Group, respectively^[7,11]

| Group tolerance in Australia | Group tolerance in Australia and Co-op zero tolerance | Co-op zero tolerance |
|---|---|--|
| Azamethiphos, azinphos-ethyl, azinphos-methyl, coumaphos, demeton, diazinon, dichlorvos, dimethoate, disulfoton, dithianon, ethion, famphur, fenclorphos, fenitrothion, fenthion, formothion, maldison ^a , methamidophos, methidathion, mevinphos, naphtalophos ^b , parathion-methyl, phosmet, pirimiphos-ethyl, pirimiphos-methyl, pyrazophos, sulprophos, temephos, tetrachlorvinphos, thiometon, S,S,S-tributylphosphorotrithioate, trichlorfon, vamidothion | Ethoprophos, fenamiphos, omethoate, phorate, prothiofos | Cadusafos, chlorfenvinphos, demeton-S-methyl, phosphamidon, tebupirimfos, terbufos |

^aISO common name is malathion.

^bWHO INN, no ISO common name available.

in food are not normally a food safety concern, consumer preference is for food that does not contain residues. Sixty-eight percent of consumers consider that reducing residue levels further than the current level is important. As a result, the Agency has developed an action plan for pesticide residue minimization with a goal of enabling consumers to make informed choices, and promoting best practice within the food industry. The overall action plan includes, among other things, development of crop specific action plans to achieve pesticide residue minimization for five priority crops: apples, pears, potatoes, tomatoes, and cereal grains.^[10]

Retailer Initiatives^[3,11,12]

Retailers may employ a range of strategies to reduce pesticide use and residues in the foods they produce and put on the market:

- Monitoring pesticide usage and residues.
- Consulting with growers, including advice on integrated pest management and on alternative pest control systems.
- Designing and providing decision tools, including crop-specific advisory sheets and frameworks for pesticide selection.
- Prohibiting or restricting the use of certain pesticides.
- Publishing monitoring results, for example, on corporate websites.
- Promoting organically grown foods.

NGO Initiatives—Ranking Residue Contents

In the United States, the Environmental Working Group has designed a report card to score pesticide residues in food products.^[13] The report card, which is based on government agency monitoring and published monitoring data, shows scores for each analyzed commodity based on a number of residue characteristics, and a combined (or total) score in these categories:

- Percentage of samples with detectable residues.
- Percentage of samples with two or more pesticides.
- Average number of pesticides found on each sample.
- Average total concentration of pesticides found.
- Maximum number of pesticides found on a single sample.
- Total number of pesticides found on a single commodity.

In the Netherlands, Natuur and Milieu (a Dutch NGO), has designed and used a ranking system based on the following components, and a calculated total score:^[14]

- For any residue not exceeding an MRL: 1 penalty point.
- For each residue of a pesticide with neurotoxic effects: 2 penalty points.



Fig. 1 European Union organic logo, available at http://europa.eu.int/comm/agriculture/qual/organic/logo/index_en.htm.

- For each residue exceeding an MRL or resulting from the use of a pesticide not authorized for use in the Netherlands (applicable also to imported foods): 4 penalty points.
- For each residue exceeding an MRL and resulting from the use of a pesticide not authorized for use in the Netherlands (applicable also to imported foods): 8 penalty points.

CONCLUSIONS

Polls have shown repeatedly that consumers are concerned about pesticide residues in food. Maximum residue limits are trading standards, which prescribe the maximum amount of particular pesticides legally permitted. These limits (MRLs) are generally based on the level of pesticides expected if good agricultural practice is followed.^[10] Other MRLs may reflect only that a pesticide is no longer authorized for use, leading to zero tolerance, and to potential problems for food exporters overseas. Since many consumers feel that current good agricultural practice is not good enough for their own or their childrens' safety, governments, retailers and NGOs have initiated actions to reduce residues. Strategies focus on production methods (integrated, organic) as well as product quality (residue-reduced or residue-free foods).

ACKNOWLEDGMENTS

Contributions from Stephanie Williamson, Pesticide Action Network U.K., are gratefully acknowledged.

REFERENCES

1. European Food Safety Authority. Risk perception and food safety: where do European consumers stand today? February 2006; http://www.efsa.eu.int/press_room/press_release/1340_en.html (accessed 21, May, 2006).
2. Food Standards Agency. Consumer concern over the use of pesticides to grow food, March 2004; <http://www.food.gov.uk/multimedia/pdfs/pestresconsumerresearch.pdf> (accessed 9 January 2005).
3. Croft D. Removing pesticides from the food chain, December 2002; <http://www.pan-uk.org/pestnews/pn58/pn58p9.htm> (accessed 19 January 2006).
4. Committee on Toxicity of Chemicals in Food, Consumer Products and the Environment. Risk assessment of mixtures of pesticides and similar substances, September 2002; <http://www.food.gov.uk/multimedia/pdfs/reportindexed.pdf> (accessed 19, January, 2006).
5. Beaumont P.; Buffin, D. A cocktail of problems, March 2002; <http://www.pan-uk.org/pestnews/pn55/pn55p10.htm> (accessed 3 March 2006).
6. Andersson, A.; Jansson A. The Swedish monitoring of pesticide residues in food of plant origin 2004. National (Swedish) Food Administration. http://www.slv.se/upload/dokument/Rapporter/Bekampningsmedel/2005/2005_17eng_Livsmedelsverket.pdf (accessed 19 January 2006).
7. Maximum residue limits—Chemical groups. Australian Food Standards No 1.4.2. <http://www.foodstandards.gov.au/foodstandardscode/> (accessed 19 January 2006).
8. World Health Organization. *Public health impact of pesticides used in agriculture*, 1990, ISBN 92 4 1561 39 4.
9. Ekstrom, G.; Hemming, H.; Palmberg, M. Swedish pesticide risk reduction 1981–1995: food residues, health hazard, and reported poisonings. *Rev. Environ. Contam. Toxicol.* **1996**, *147*, 119–147.
10. Food Safety Authority. Progress on an agency action plan to minimize pesticide residues in food, May 2004; <http://www.food.gov.uk/multimedia/pdfs/fsa040502.pdf> (accessed 19 January 2006).
11. The Co-operative Group. Co-op and the responsible use of pesticides. Consumer issues website. <http://www.co-op.co.uk> (accessed 27 February 2006).
12. Barker K. Co-operative retail's pesticide reduction program, July 2005; <http://www.pan-europe.info/conferences/PURE%20workshop%202005/Kevin%20Barker%20Co-op.pdf> (accessed 2, March, 2006).
13. Environmental Working Group. Report card: pesticides in produce 2005; <http://www.foodnews.org/reportcard.php> (accessed 20 December 2005).
14. Muilerman, H. Dutch supermarket residue campaign. *Pestic. News* **2006**, *71*, 6–7.

Cowpea Insects: Ecology and Control

Keyan Zhu-Salzman

Department of Entomology, Texas A&M University, College Station, Texas, U.S.A.

Larry L. Murdock

Department of Entomology, Purdue University, West Lafayette, Indiana, U.S.A.

INTRODUCTION

Of African origin, cowpea (*Vigna unguiculata* L. Walp) is one of the most ancient crops. This grain legume provides high-quality protein for humans and livestock. Numerous herbivorous insects attack cowpea, often resulting in severely reduced yields. In the long history of battling insect pests, scientists have accumulated a great diversity of cowpea accessions from around the world with different susceptibilities to herbivores. The geographic distribution of the hosts, in turn, has influenced the composition of the pest complex and the insects' feeding habits, vulnerability to parasitoids and predators, movement patterns, and oviposition behavior. Research has been conducted on the molecular bases of plant defense and insect counter defense, environmental influences on the genetic architecture of both parties, as well as on developing effective pest control practices. This article summarizes current progress with cowpea, and gives implications for pest management.

COWPEA

Cowpea, the most economically important indigenous legume crop of Africa, thrives in low-to-moderate rainfall zones from Senegal in the west of the continent to Sudan and Kenya in the east, and to Botswana and Mozambique in the south. There is substantial production also in Brazil and U.S.A., as well as in southern Europe and Asia.^[1]

Cowpea grain is rich in easily digestible protein and carbohydrate. Total energy content is nearly that of cereal grains. As a species, cowpea is highly variable in overall plant size, growth habit, and size, color, and texture of the leaves and seeds. Large seed size, rough and crack-free surface, mild taste, and lack of bruchid holes are features favored by cowpea consumers. Young leaves, immature pods, and fresh green seeds are used as food and cowpea hay is used as livestock feed.

Cowpea plants are tolerant to heat, drought, and poor soil conditions, and fix atmospheric nitrogen. The deep root system helps stabilize the soil, and the ground cover it provides preserves moisture; these

traits are particularly important in the drier regions, where moisture is at a premium and the soil is fragile and subject to wind erosion.

INSECT PESTS

Lepidoptera

The most important lepidopteran pest of cowpea is the legume pod borer (*Maruca vitrata*); its larvae attack growing shoot tips, flowers, and developing pods. It is found in the tropics throughout the world. Other lepidopteran pests include the cowpea seed moth (*Cydia ptychora*) in southern Nigeria, the lesser corn-stalk borer (*Elasmopalpus lignosellus*) in the Americas, and *Heliothis* and *Spodoptera* species in the Americas and tropical Africa.

Coleoptera

Adults and larvae of the cowpea curculio (*Chalcoedermus aeneus*) attack developing seeds and cause major losses. The most serious postharvest pest of cowpea is the cowpea bruchid, *Callosobruchus maculatus*. The fast developmental time (three to four weeks per generation) and high reproductive capacity (40–60 eggs per female) of *C. maculatus* cause populations to expand quickly such that the harvested grain may be destroyed within a few months. Numerous other Coleoptera are minor or sporadic pests of cowpea in the field or in storage.

Heteroptera

Pod-sucking bugs make up a complex of insects that cause serious damage. In Africa, the coreids *Clavigralla tomentosicollis* and *Riptortus dentipes* are the most important species. In the southeastern U.S.A., the southern green stinkbug *Nezara viridula*, a pentatomid, is the major bug pest, while in California it is *Lygus hesperus* of the family Miridae. In Latin America, various coreids and pentatomids make up a yield-reducing pest complex. Leafhoppers are serious

pests in tropical and subtropical cowpea-growing areas, as are aphids, particularly the black cowpea aphid, *Aphis craccivora*. This worldwide species can occur in huge numbers and kill plants, though their more important role may be to transmit plant diseases.

Thysanoptera

Flower thrips are counted among the worst and most widespread pests of cowpea in Africa, and may cause complete loss of grain yield. Nymphs and adults may damage the terminal leaf buds and bracts/stipules, but the most severe damage results from feeding on the flower buds and the flowers themselves.

PEST MANAGEMENT

Losses of cowpea grain production can be 90% or higher. Current insect-control practices on cowpea involve: 1) chemical pesticides; 2) cultural procedures such as crop rotation and intercropping with a cereal; 3) biological agents such as neem and other botanical preparations; and 4) resistant plant varieties. Chemical control through sprays, dusts, and fumigation is often highly effective. Several chemical insecticides are used for cowpea insect pests in the developed world as well as in Africa. Pyrethroids like cypermethrin or the organophosphate dimethoate, typically sprayed at flowering and podding stages, give excellent control. Chemical pesticides, however, pose threats to human health and the environment, and in Africa are often not available or are too expensive for resource-poor farmers to use. Natural enemies and crop rotations help suppress pest insect populations. Several low-cost and environment-friendly storage technologies have been developed to counter postharvest losses, such as solar disinfestation and hermetic storage in plastic bags.^[2]

Improved cowpea cultivars that resist or tolerate biotic and abiotic stresses are highly desirable because they enhance yield and quality of the grain with little or no additional inputs. Approximately 20,000 cowpea accessions have been collected from around the globe; these germplasm collections have been a key to cowpea improvement through breeding. High-yielding and short-season varieties have been developed. Backcrossing has helped combine resistance to several pests and diseases with improved seed quality and high-yielding traits. Several early maturing cowpea cultivars have been developed with resistance to cowpea aphid, cowpea curculio, root-knot nematodes, and cowpea bruchid as well as the parasitic weed *Striga gesnerioides*.^[3] Marker-assisted selection technology is in the process of being adopted in cowpea breeding programs.^[4] Genetic transformation of cowpea to

introduce new sources of insect resistance is an ongoing effort and may deliver new sources of resistance against insects not attainable through conventional breeding, e.g., *Bacillus thuringiensis* cowpea.

PLANT-INSECT INTERACTIONS AND ECOLOGICAL IMPACT

Plant resistance to insects is known to be genetically variable and often controlled by quantitative trait loci. Insects, having coexisted with plants for millions of years, have developed mechanisms to cope with various plant toxins and antinutrient factors. The evolutionary interaction between plant hosts and their herbivores in a specific ecological context is clearly reflected by the diversity of cowpea and cowpea bruchid populations. A landrace of cowpea (TVu2027) produces seeds resistant to cowpea bruchid.^[5] Certain populations of cowpea bruchid, in turn, have been found that overcome the TVu2027 resistance.^[6]

Ecological divergence of cowpea plants has affected spatial distribution and oviposition behavior of insect herbivores, as illustrated in a study where Asian and African cowpea bruchid populations were compared.^[7] Furthermore, expression of the alleles controlling distinct behaviors is clearly environment dependent. While this evolutionary adaptation is reflected among different ecological populations, it is interesting that insects from a single population can alter their gene expression when confronted by plant defenses. To counter dietary protease inhibitors, for example, the cowpea bruchid reconfigures its digestive transcriptome to minimize the impact of the dietary challenges.^[8] This likely occurs in nature when insects expand their range or switch to new hosts.

Predators, parasitoids, and pathogens, as well as other herbivores affect the target insect and are integral factors in composing a strategy for pest suppression. Too often, experimental studies focus on only one natural enemy species. However, attention has been increasingly paid to the effects of a multiple natural enemy-pest assemblages. Coexistence of cowpea aphids with pea aphids decreased control of the pea aphids by a specialist parasitoid, but incorporating a generalist predator resulted in improved pest control.^[9] The dynamics of natural enemy guilds can also be altered by the cropping systems adopted, as exemplified in effects of cowpea mono- and polycultures on predatory arthropods.^[10]

CONCLUSIONS

To formulate an effective, low-cost, and ecofriendly pest management strategy, good background knowledge regarding the host plant and the pest insects

and their environment is essential. Research-based technology will lead to a better understanding of the genetic architecture of population dynamics and the molecular bases of plant defense and insect counter-defense mechanisms. It will also facilitate development of cowpea varieties combining pest resistance and desired grain-quality traits that are suitable for particular agroecosystems. A rationalized pest management plan that takes into account both biotic and abiotic factors is necessary to increase the sustainability of production systems.

REFERENCES

1. Langyintuo, A.S.; Lowenberg-DeBoer, J.; Faye, M.; Lambert, D.; Ibro, G.; Moussa, B.; Kergna, A.; Kushwaha, S.; Musa, S.; Ntougam, G. Cowpea supply and demand in west and central africa. *Field Crops Res.* **2003**, *82*, 215–231.
2. Murdock, L.L.; Seck, D.; Ntougam, G.; Kitch, L.; Shade, R.E. Preservation of cowpea grain in sub-Saharan Africa—bean/cowpea CRSP contributions. *Field Crops Res.* **2003**, *82*, 169–178.
3. Ehlers, J.D.; Hall, A.E. Cowpea (*Vigna unguiculata* L. Walp). *Field Crops Res.* **1997**, *53*, 187–204.
4. Ouedraogo, J.T.; Gowda, B.S.; Jean, M.; Close, T.J.; Ehlers, J.D.; Hall, A.E.; Gillaspie, A.G.; Roberts, P.A.; Ismail, A.M.; Bruening, G.; Gepts, P.; Timko, M.P.; Belzile, F.J. An improved genetic map for cowpea (*Vigna unguiculata* L.) combining AFLP, RFLP, RAPD and biochemical markers and biological resistance traits. *Genome* **2002**, *45*, 175–188.
5. Singh, S.R. Cowpea cultivars resistant to insect pests in world germplasm collection. *Trop. Grain Legume Bull.* **1977**, *9*, 1–7.
6. Shade, R.E.; Murdock, L.L.; Kitch, L.W. Interactions between cowpea weevil (coleoptera: bruchidae) populations and vigna (leguminosae) populations. *J. Econ. Entomol.* **1999**, *92*, 740–745.
7. Fox, C.W.; Stillwell, R.C.; Amarillo, A.R.; Czesak, M.E.; Messina, F.J. Genetic architecture of population differences in oviposition behaviour of the seed beetle *Callosobruchus maculatus*. *J. Evol. Biol.* **2004**, *17*, 1141–1151.
8. Zhu-Salzman, K.; Koiwa, H.; Salzman, R.A.; Shade, R.E.; Ahn, J.-E. Cowpea bruchid *Callosobruchus maculatus* uses a three-component strategy to overcome a plant defensive cysteine protease inhibitor. *Insect Mol. Biol.* **2003**, *12*, 135–145.
9. Cardinale, B.J.; Harvey, C.T.; Gross, K.; Ives, A.R. Biodiversity and biocontrol: emergent impacts of a multi-enemy assemblage on pest suppression and crop yield in an agroecosystem. *Ecol. Lett.* **2003**, *6*, 857–865.
10. Nampala, P.; Adipala, E.; Ogenga-Latigo, M.W.; Kyamanywa, S.; Obuo, J.E. Effect of cowpea monocultures and polycultures with sorghum and greengram on predatory arthropods. *Ann. Appl. Biol.* **1999**, *135*, 457–461.

Crop Insect Control: Ant Roles

E. Rolando Lopez-Gutierrez

Clemson University, Charleston, South Carolina, U.S.A.

Gloria S. McCutcheon

Coastal Research and Education Center, Clemson University, Charleston, South Carolina, U.S.A.

INTRODUCTION

Ants (Hymenoptera: Formicidae) are among the most widespread and abundant of insects. They thrive in various environments over a diverse geographic range. Several characteristics of ants make them attractive potential candidates for use in biological control of insect pests.

A detailed knowledge of the behavior of an ant species in a particular environment, taking into account their interactions with the other ant species sharing the same area, has proven to be instrumental in successful pest management.

PROMISING ATTRIBUTES OF ANTS FOR PEST CONTROL

Collectively, ants rival many more visible fauna in geographic range and sometimes in local biomass.^[1,2] Different ant species occupy a wide range of environments, as well as a variety of microhabitats within particular ecosystems.^[2-7] Important attributes of beneficial ants have been summarized by Risch and Carroll.^[8] The feeding behavior of various ant species and their potential food web impacts are well documented.^[3,4,6,7,9,10] Many species are highly predatory, and their invertebrate prey includes adults, larvae, and eggs of numerous crop insect pests,^[11] including economically damaging *Orthopteran*, *Homopteran*, *Hemipteran*, *Dipteran*, and *Lepidopteran* species. Nevertheless, the value of ants as biological control agents is often underestimated, particularly in cases of egg predation.^[4,7,8,12] Numerous early studies of ant foraging ecology are helpful in gauging the potential role of these insects in pest management; these are summarized in two major review papers.^[3,13]

The social nature of ants leads to unique advantages for use in controlling insect pests. Because of their ability to recruit nestmates to recently encountered food sources, they are often able to outcompete non-social predators on patches of prey, leading to rapid control of a pest.^[3,4,7] Their wide-ranging foraging capacity, as

well as directed searching through communication among workers from a given nest, can be effective at locating and overcoming very low densities of prey: *Oecophylla longinoda* has been shown to eliminate pest densities as low as one to two adult coconut bugs, *Pseudotheraptus wayi* Brown (Bruwer, 1992) or *Pseudotheraptus devastans* (Distant), per coconut tree in African plantations.^[7]

SUCCESS IN PEST CONTROL

Ants have proven important in keeping insect pests below nuisance or economic injury levels in a variety of agricultural and other settings. This occurred in some cases with little or no intentional human intervention (Fig. 1). Pimentel^[14] and Pimentel and Uhler^[16] have reported on spontaneous control of houseflies by ants in Hawaii, Puerto Rico, and the Philippines by *Pheidole megacephala* Fabricius, *Solenopsis geminata* (F.), and *Pheidologeton affinis* (Jerdon, 1851), respectively. Previously considered a scavenger, *Lasius neoniger* Emery feeds on eggs and larvae of *Agrotis ipsilon* (Hufnagel, 1766) in turfgrass in the United States and Canada.^[10,12] Additional examples of naturally occurring pest suppression by ants are documented in McCutcheon and Ruberson,^[13] Perfecto and Castiñeiras,^[4] and Way and Khoo.^[7]

Ants have also been manipulated intentionally by agriculturalists who have taken advantage of the feasibility of multiplying ant nests and moving them about according to particular pest management needs. The oldest known case of purposeful ant nest manipulation by humans is that of *Oecophylla smaragdina* (Fabricius) in Asia.^[4,7] Nests of these ants are relatively easy to locate and transport because they are constructed in the canopies of trees rather than the below ground. Farmers also provide the ants with supplemental protein in the form of discarded seafood scraps during periods when prey populations are low. *O. smaragdina* is helpful primarily in controlling citrus pests and has also been used against *Amblypelta* spp. (Hemiptera: Coreiidae) in coconut plantations.^[4,7] Other cases of



Fig. 1 *S. invicta* attacking imported cabbageworm.

ant management by farmers are those of *Dolichoderus toracicus* Smith in cocoa farms in Indonesia and Malaysia^[4,7] and of *Azteca chartifex* Forel in Brazil,^[4] where proliferation of ant nests in desired field locations is accomplished through provision of dead palm fronds as nesting material. A recent success in fostering predatory ant populations is that of *P. megacephala* (Fabricius, 1793) in Cuba, where field workers have learned to facilitate the establishment of new colonies in artificial settings by providing food and nest material in the form of table scraps and molasses sandwiched within tied stacks of banana leaves. Once colonized by *P. megacephala*, the bundles are transported

to agricultural fields where the ants effectively control crop pests such as banana weevils *Cosmopolites sordidus* (Germ.) and sweet potato weevils *Cylas formicarius elegantulus* (Summer).^[4,16] To favor particular ant species, it has been necessary in each case to discover the specific elements important to their survival.

MANAGEMENT COMPLEXITY

Ants typically compete for territorial dominance against other ant species within the same ecosystem,^[1,2,17,18] and the outcomes of these encounters can be either beneficial or damaging to human interests. Understanding these interactions is important when attempting to favor a given ant species.^[7] Crops are grown in a wide range of ecological conditions from much simplified arable fields through arboreal monocultures to complex systems approaching mature forest.^[7,18–20] Purposely or inadvertently, managers alter vegetation complexity and, in this way, influence competitive relations among ant species, as illustrated by recent changes in coffee plantation management practices^[20] and restoration management of longleaf pine *Pinus palustris* ecosystems in the Southeastern United States.^[19] In working with potentially undesirable ants, it can be helpful to recognize that, often, “the worst enemy of an ant is another ant,”^[2,21] and to proceed accordingly in control efforts.^[2,10,17,18] One

Table 1 Beneficial ant–crop interactions

| Ant species | Common name | Agroecosystem | Pest insects | Region of observations | Human involvement | References |
|---|-----------------------|---|--|---|---|------------|
| <i>Oecophylla smaragdina</i> (Fabricius) | Weaver ant | Orchards | Citrus, coconut, and cocoa pests | Asia, Australia | Active management (e.g., multiplication, movement, and feeding of colonies) | [4,6] |
| <i>Pheidole megacephala</i> (Fabricius, 1793) | Big-headed ant | Field crops and orchards: banana, coconut, shade coffee | Sweet potato weevil, black cutworm, banana weevil, coreids | Tropics | Accidental introductions; active management (e.g., multiplication, movement, and feeding of colonies) | [4,7,16] |
| <i>Solenopsis invicta</i> Buren, 1972 | Red imported fire ant | Field crops, turfgrass | Fall army worm, corn earworm, boll weevil | Tropical America, Southeastern United States | Accidental introduction | [4,7,20] |
| <i>Iridomyrmex humilis</i> (Mayr, 1968) | Argentine ant | Fruit orchards, turfgrass | Black cutworm, yellow jackets | Western United States | Accidental introduction | [7,18] |
| <i>Anoplolepis gracilipes</i> (Fr. Smith) | Crazy ant | Orchards | Pests of coconut | Pacific Islands | Accidental introduction | [7] |
| <i>Wasmannia auropunctata</i> (Roger) | Little fire ant | Citrus groves and other orchards | Pests of citrus and coconut | Tropical America, Africa, Florida, California | Accidental introduction | [7] |



Fig. 2 *L. neoniger* and honeydew-producing aphid.

example of promising research using a combined approach to a problematic ant involves the biological control of *Solenopsis invicta* in the southeastern United States.^[22] Except for *O. smaragdina*, the ant species included in Table 1 are considered invasive in areas where they have been introduced. The movement of ants outside their native ranges for the purpose of biological control is not endorsed here due to the high risk and unpredictable consequences of such introductions.^[21]

CONCLUSION

The examples cited above indicate potentially greater roles for ants in biological control, but additional studies within the range of relevant settings will be needed before wider practical applications are possible. Many predatory species can present beneficial and/or problematic attributes depending on the circumstances (Refs.^[21] and Table 1). In the past, the use of ants to control pest populations has been discouraged on the basis of two general concerns: 1) ants commonly favor honeydew-producing *Homopteran* that themselves can become serious pests^[21,23] (Fig. 2); and 2) foraging by generalist ant species may inflict strong mortality on beneficial species.^[7,21] Detailed knowledge of individual ecosystems is required to ascertain and adjust the balance between possible benefits of ants in controlling particular pests and any potential undesirable effects.

ACKNOWLEDGMENTS

We want to thank Dr. Linda C. Roth (Department of Forestry and Natural Resources, Clemson University) for her very pertinent criticisms on an early version of

the manuscript. We also thank Mark Schaffer (Coastal Research and Education Center) for assisting with the graphics.

ARTICLES OF FURTHER INTEREST

Red Imported Fire Ant in Crop Insect Control, p. 113.

REFERENCES

- Hölldobler, B.; Wilson, E.O. *The Ants*; Belknap Press of the Harvard University Press: Cambridge, MA, 1990.
- Hölldobler, B.; Wilson, E.O. *Journey to the Ants—A Story of Scientific Exploration*; Belknap Press of the Harvard University Press: Cambridge, MA, 1994.
- Carroll, C.R.; Janzen, D.H. Ecology of foraging by ants. *Annu. Rev. Ecol. Syst.* **1973**, *4*, 231–257.
- Perfecto, I.; Castiñeiras, A. Deployment of predaceous ants and their conservation in agroecosystems. In *Conservation Biological Control*; Barbosa, P., Ed.; Academic Press: New York, 1998; 269–289.
- Smith, M.R. House-infesting ants of the eastern United States. Their recognition, biology, and economic importance. In *USDA-ARS Technical Bulletin No. 1326*; U.S. Gov. Printing Office: Washington, DC, 1965.
- Torres, J.A. Niches and coexistence of ants in Puerto Rico: repeated patterns. *Biotropica* **1984**, *16*, 284–295.
- Way, M.J.; Khoo, K.C. Role of ants in pest management. *Annu. Rev. Entomol.* **1992**, *37*, 479–503.
- Risch, S.J.; Carroll, R. The ecological role of ants in two Mexican agroecosystems. *Oecologia* **1982**, *55*, 114–119.
- Ayre, G.L. Laboratory studies on the feeding habits of seven species of ants (Hymenoptera: Formicidae) in Ontario. *Can. Entomol.* **1963**, *95*, 712–715.
- López, R.; Held, D.W.; Potter, D.A. Management of a mound-building ant, *Lasius neoniger* Emery, on golf putting greens and tees using delayed-action baits or Fipronil. *Crop Sci.* **2000**, *40*, 511–517.
- Risch, S.J.; Carroll, R. Effect of a keystone predaceous ant, *Solenopsis geminata*, on arthropods in a tropical agroecosystems. *Ecology* **1982**, *63*, 1979–1983.
- López, R.; Potter, D. Ant predation on eggs and larvae of the black cutworm (Lepidoptera: Noctuidae) and Japanese beetle (Coleoptera: Scarabaeidae) in turfgrass. *Environ. Entomol.* **2000**, *29*, 116–125.
- Traniello, J.F.A. Foraging strategies of ants. *Annu. Rev. Entomol.* **1989**, *34*, 191–210.
- Pimentel, D. Relationship of ants to fly control in Puerto Rico. *J. Econ. Entomol.* **1955**, *48*, 28–30.
- Pimentel, D.; Uhler, L. Ants and the control of houseflies in the Philippines. *J. Econ. Entomol.* **1969**, *62*, 248.
- Oppenheim, S. Alternative agriculture in Cuba. *Am. Entomol.* **2001**, *47*, 216–227.
- Bhatkar, A.; Whitcomb, W.H.; Buren, W.F.; Callahan, P.; Carlyse, T. Confrontation behavior between *Lasius*

- neoniger* (Hymenoptera: Formicidae) and the imported fire ant. *Environ. Entomol.* **1972**, *1*, 274–279.
18. Fluker, S.S.; Beardsley, J.W. Sympatric associations of three ants: *Iridomyrmex humilis*, *Pheidole megacephala*, and *Anoplolepis longipes* in Hawaii. *Ann. Entomol. Soc. Am.* **1970**, *63*, 1290–1296.
 19. Lubertazzi, D.; Tschinkel, W.R. Ant community change across a ground vegetation gradient in North Florida's longleaf pine Flatwoods. *J. Insect Sci.* **2003**, *3*, 1–17.
 20. Perfecto, I.; Vandermeer, J.H. Microclimatic changes and the indirect loss of ant diversity in a tropical agroecosystems. *Oecologia* **1996**, *108*, 577–583.
 21. Holway, D.A.; Lach, L.; Suarez, A.V.; Tsutsui, N.D.; Case, T.J. The causes and consequences of ant invasions. *Annu. Rev. Ecol. Syst.* **2002**, *33*, 181–233.
 22. Williams, D.F.; Oi, D.H.; Porter, D.P.; Pereira, R.M.; Soriano, J.A. Biological control of imported fire ants. (Hymenoptera: Formicidae). *Am. Entomol.* **2003**, *49*, 150–163.
 23. Reimer, N.J.; Beardsley, J.W. Effectiveness of Hydramethylnon and Fenoxycarb for the control of the big-headed ant (Hymenoptera: Formicidae), an ant associated with mealybug wilt of pineapple in Hawaii. *J. Econ. Entomol.* **1990**, *83*, 74–84.

Crop Insect Control: Red Imported Fire Ants

Gloria S. McCutcheon

Clemson University, Charleston, South Carolina, U.S.A.

John Ruberson

*Department of Entomology, University of Georgia,
Tifton, Georgia, U.S.A.*

INTRODUCTION

Ants may be herbivorous and/or carnivorous—consuming plant exudates, seeds, and living or dead plant and animal matter. In addition, ants, unlike most other agricultural denizens, manipulate the physical environment and alter their habitat. Therefore ants can be important in shaping agroecosystems in which they occur.

BENEFICIAL ROLE

There are 9000 described species of ants worldwide.^[1] A number of these species frequent agroecosystems and can play significant roles in them.^[2] The ecological niche of ants in agroecosystems is complex, depending on soil type,^[3] climate, nesting strategies, diet, life spans, diel activity patterns, crop plants (including availability of nectar), cropping practices, and the fauna of the system.^[4] Way and Khoo^[2] reviewed the role of ants in agroecosystems and highlight several ant taxa important in agriculture. The positive value of ants as predators has long been acknowledged. The aggressive ant *Oecophylla smaragdina* F. appears to have been manipulated by the Chinese to manage citrus pests as early as A.D. 304 and is the first known example of an arthropod natural enemy propagated and sold.^[5] *Oecophylla longinoda* occurs in Africa, and *O. smaragdina* in Asia and Australia, where they are useful predators in cocoa, citrus, coffee, oil palm, mango, coconut, and timber production.^[2]

An agriculturally important ant genus in the New World is *Solenopsis*. The red imported fire ant, *Solenopsis invicta* Buren (hereafter referred to as “fire ant”), and other congeners can be very important predators in Neotropical and Nearctic agricultural systems. This species pervades many agricultural systems and provides an excellent example of the complex role of ants in agriculture (Fig. 1).

S. invicta is the dominant predaceous arthropod in cotton in many areas across the southern United States, having displaced the native fire ant, *Solenopsis*

geminata (F.), in most areas. Predation of boll weevil larvae, *Anthonomus grandis* (Boheman), by *S. invicta* can be substantial, significantly reducing weevil damage to cotton in Texas.^[6] *S. invicta* was one of the dominant predators of heliothine eggs in Georgia cotton.^[7] It was the most abundant predator in a conservation tillage study in South Carolina.^[8] Ants were more abundant in no-till cotton doublecropped with rye than in cotton that had been disked or was monocropped (disked or no-till). In Alabama cotton, *S. invicta* was a very abundant predator and was negatively correlated with 16 herbivorous taxa, including lepidopterous larvae and several hemipterous pests.^[9] Lepidopterous larvae did not reach economic thresholds when fire ants were abundant.

S. invicta affects several soybean insect pests in the southern United States, and it can recruit rapidly, allowing it to attack arthropods of varying sizes. Populations of the southern green stink bug, *Nezara viridula* (L.), and threecornered alfalfa hopper, *Spissistilus festinus* (Say), are reduced by *S. invicta* in Alabama soybeans.^[9] In South Carolina, researchers^[10] observed low populations of *S. invicta* in the canopy of doublecropped soybean, although mounds were abundant. Where fire ants were increased by conservation tillage, *S. festinus*, *N. viridula*, and velvetbean caterpillar, *Anticarsia gemmatilis* Hübner did not reach economic threshold levels, and soybean yield was not reduced. In Louisiana, *S. invicta* accounted for 52% to 95% of the predation of *A. gemmatilis* pupae under the soil surface in soybean.^[11]

Predation by *S. invicta* is important in sugarcane production. In Florida and in Louisiana, *S. invicta* is an important integrated pest management (IPM) tool for control of the sugarcane borer, in combination with other control tactics including plant resistance^[12] and insecticides.^[13] *S. invicta* also plays a key role in sugarcane borer suppression in Brazil.^[14]

Other examples of a positive role for fire ants in pest management include sweet sorghum, maize, cucumber, lantana and alfalfa. *S. invicta* suppressed fall armyworm, *Spodoptera frugiperda* (J.E. Smith), populations in sweet sorghum in Louisiana^[15] and in maize



Fig. 1 Fire ants with brood.

in Nicaragua, where corn leafhopper, *Dalbulus maidis* (DeLong and Walcott) was also reduced.^[16] In cucumber, *S. invicta* was a significant predator of pickleworm pupae in South Carolina.^[17] Fire ants predate a variety of arthropods, including significant numbers of greenhouse whiteflies, *Trialeurodes vaporariorum* (Westwood), on lantana in the greenhouse. They also consume alfalfa weevil larvae, *Hypera postica* (Gyllenhal), in greenhouse-grown alfalfa.^[18]

NEGATIVE EFFECTS IN CROPPING SYSTEMS

Both the *S. invicta* mounds and their inhabitants can be problematic in cropping systems. *S. invicta* nests in mounds with interconnected galleries and chambers extending 30–40 cm down and radiating outward 5–10 m just below the ground surface.^[19] Fire ant mounds interfere with farm machinery and contribute to unsightly landscapes. The ants themselves can decrease crop yields directly and indirectly. Fire ants have been reported to destroy seeds, root systems, and seedlings. Ants feeding at the base of the fruit or around the plant (girdling) can induce premature fruit shed. Indirectly, *S. invicta* can reduce yield by attacking important pollinators that nest in the ground or by tending homopterans.

Like many ants, *S. invicta* often associates with Homoptera and can contribute to increased populations of aphids, whiteflies, scales, and mealybugs (e.g., Ref. ^[20]), some of which may vector phytopathogens. Ants benefit by obtaining carbohydrates in the form of honeydew and can readily acquire protein by predated the homopterans as needed. Predators of the ant-protected Homoptera often suffer population reductions as a result of the ant's protective behavior. In pecans, *S. invicta* associates with the mealybug, *Dysmicoccus*

morrisoni (Hollinger), covering mealybug colonies on callus tissue with soil and debris, defending the colony, and collecting honeydew.^[21] Cotton aphid, *Aphis gossypii* Glover, populations can increase when fire ants are abundant in cotton.^[22] Cotton under conservation tillage has larger fire ant populations and often experiences larger cotton aphid populations as a result.^[23] When aphid populations are high in cotton, predation on lepidopteran eggs by fire ants and other predators declines.

IMPACT ON PREDACEOUS ARTHROPODS

Aggressive ant species may displace other ants present in agroecosystems. In Florida sugarcane fields, *S. invicta* populations reduced relative abundance of other species of ants.^[24]

Fire ants prey on natural enemies of many of the pest arthropods in crop systems. They prey on predaceous arthropods of all life stages as well as parasitized pest eggs, aphids, and parasitic insect pupae. Predation on parasitized pests destroys immature parasitoids developing inside the eggs, thereby impairing effective biological control. For example, parasitism of heliothine eggs and cotton aphids in cotton can be as high as 70% in some areas of the U.S. Cotton Belt. *S. invicta* preys on cocoons of the braconid wasp, *Cardiochiles nigriceps* Viereck, an important parasitoid of the tobacco budworm, *Heliothis virescens* (F.), in tobacco, cotton, soybean, pigeon pea, tomato, and many other crops.^[25] In South Carolina, increased densities of *S. invicta* in conservation tillage cotton resulted in decreased densities of several other important predators. Conditions that favor ants often result in decreased densities of other major natural enemies.^[8]

S. invicta tends aphids and protects them from other important predators. Thus a mutualistic relationship exists in which aphids provide honeydew for ant nutrition and ants provide protection from aphid predators.

CONCLUSION

The role of ants in agricultural systems remains poorly understood. The social behavior of ants, their shifting nutritional demands and omnivory, and their variable interactions with cropping practices and crops have made evaluations of ant impacts very difficult. Ants interact with species at all levels of the food chain, the producers, the consumers, and the carnivores. While pest managers may benefit from ant predation when herbivores are the principal food, ant herbivory and destruction of arthropod natural enemies may lead to substantial crop damage. Understanding the outcomes of ant interactions with crop systems,

herbivores, and entomphagous species will be critical for integrating ants into pest management schemes more effectively.

ACKNOWLEDGMENTS

We thank Winfield Sterling for the photograph and Carol Ferguson for her willing assistance in the preparation of this manuscript.

REFERENCES

- Boltin, B. *Identification Guide to the Ant Genera of the World*; Harvard University Press: Cambridge, MA, 1994.
- Way, M.J.; Khoo, K.C. Role of ants in pest management. *Annu. Rev. Entomol.* **1992**, *37*, 479–503.
- Johnson, R.A. Soil texture as an influence on the distribution of the desert seed-harvester ants *Pogonomyrmex rugosus* and *Messor pergandei*. *Oecologia* **1992**, *89*, 118–124.
- Kaspari, M.A. Primer on ant ecology. In *Ants: Standard Methods for Measuring and Monitoring Biodiversity*; Agosti, D., Majer, J., Alonso, L., Schultz, T.R., Eds.; The Smithsonian Institution Press: Washington, DC, 2000; 9–24.
- Huang, H.T.; Yang, P. The ancient cultured citrus ant. *BioScience* **1987**, *37* (9), 665–670.
- Jones, D.; Sterling, W.L. Manipulation of red imported fire ants in a trap crop for boll weevil suppression. *Environ. Entomol.* **1979**, *8*, 1073–1077.
- Ruberson, J.R.; Greenstone, M.H. Predators of budworm/bollworm eggs in cotton: an immunological study. Proceedings of the Beltwide Cotton Conferences, Cotton Insect Research and Control Conference, San Diego, CA, Jan 5–9, 1998; Dugger, P., Richter, D., Eds.; National Cotton Council: Memphis, TN, 1998, 1095–1098.
- McCutcheon, G.S. Effect of surface tillage on population dynamics of beneficial arthropods in cotton. Proceedings of the Beltwide Cotton Conferences, Orlando, FL, Jan 3–7, 1999; Dugger, P., Richter, D., Eds.; National Cotton Council: Memphis, TN, 1999, 1117–1120.
- Eubanks, M.D. Estimates of the direct and indirect effects of red imported fire ants on biological control in field crops. *Biol. Control* **2001**, *21*, 35–43.
- <http://cipm.ncsu.edu/cipmprojects/Reports/00Reports1/MANLEYDG2000S.html> (accessed September 2003).
- Lee, J.; Johnson, S.J.; Wright, V.L. Quantitative survivorship analysis of the velvetbean caterpillar (Lepidoptera: Noctuidae) pupae in soybean fields in Louisiana. *Environ. Entomol.* **1990**, *19*, 978–986.
- Bessin, R.T.; Reagan, T.E. Cultivar resistance and arthropod predation of sugarcane borer (Lepidoptera: Pyralidae) affects incidence of deadhearts in Louisiana sugarcane. *J. Econ. Entomol.* **1993**, *86* (3), 929–932.
- Adams, C.T.; Summers, T.E.; Lofgren, C.S.; Focks, D.A.; Prewitt, J.C. Interrelationship of ants and the sugarcane borer in Florida sugarcane fields. *Environ. Entomol.* **1981**, *10*, 415–418.
- Rossi, M.N.; Fowler, H.G. Ant predation of larval *Diatraea saccharalis* Fab. (Lep., Crambidae) in new sugarcane in Brazil. *J. Appl. Entomol.* **2000**, *124* (5), 245–247.
- Fuller, B.W.; Reagan, T.E.; Flynn, J.L.; Boetel, M.A. Predation on fall armyworm (Lepidoptera: Noctuidae) in sweet sorghum. *J. Agric. Entomol.* **1997**, *14* (2), 151–155.
- Perfecto, I. Ants (Hymenoptera: *Formicidae*) as natural control agents of pests in irrigated maize in Nicaragua. *J. Econ. Entomol.* **1991**, *84* (1), 65–70.
- Eley, K.D. Pickleworm: mortality on cucumbers in the field. *Environ. Entomol.* **1980**, *9*, 806–809.
- Morrill, W.L. Red imported fire ant predation on the alfalfa weevil and pea aphid. *J. Econ. Entomol.* **1978**, *71*, 867–868.
- Vinson, S.B. Invasion of the red imported fire ant. *Am. Entomol.* **1997**, *43*, 23–39.
- Helms, K.R.; Vinson, S.B. Widespread association of the invasive ant *Solenopsis invicta* with an invasive mealybug. *Ecology* **2002**, *83* (9), 2425–2438.
- Tedders, W.L.; Reilly, C.C.; Wood, B.W.; Morrison, R.K.; Lofgren, C.S. Behavior of *Solenopsis invicta* (Hymenoptera: Formicidae) in pecan orchards. *Environ. Entomol.* **1990**, *19*, 44–53.
- Reilly, J.J.; Sterling, W.L. Interspecific association between the red imported fire ant (Hymenoptera: Formicidae), aphids, and some predaceous insects in a cotton agroecosystem. *Environ. Entomol.* **1983**, *12*, 541–545.
- Lewis, W.J.; Haney, P.B.; Reed, R.; Walker, A. A total systems approach for sustainable cotton production in Georgia and the Southeast: first-year results. Proceedings of the Beltwide Cotton Conferences, Cotton Insect Research and Control Conference, New Orleans, LA, Jan 6–10, 1997; Drugger, P., Richter, D., Eds.; National Cotton Council: Memphis, TN, 1997, 1129–1134.
- Cherry, R.H.; Nuessly, G.S. Distribution and abundance of imported fire ants in Florida sugarcane fields. *Environ. Entomol.* **1992**, *21*, 767–770.
- Lopez, J.D., Jr. Emergence pattern of an overwintering population of *Cardiochiles nigriceps* in Central Texas. *Environ. Entomol.* **1982**, *11*, 838–842.

Crop Losses to Animal Pests, Plant Pathogens, and Weeds

E.-C. Oerke

Institute for Plant Diseases, Rheinische Friedrich-Wilhelms-Universität Bonn,
Bonn, Germany

Coco-Field

INTRODUCTION

Growers have to compete with animal pests (e.g., insects, mites, rodents, slugs and snails, and birds), plant pathogens (viruses, bacteria, fungi, chromista, and nematodes), and weeds (i.e., competitive plants)—collectively called pests—for the crop products grown for human purposes. They may be controlled by physical (cultivation, mechanical weeding, etc.), biological (cultivar choice, crop rotation, antagonists, predators, etc.) and chemical measures. According to Zadoks and Schein^[1] various levels of yield losses may be differentiated, e.g., primary and secondary losses and direct and indirect losses; according to this, pests not only threaten crop productivity and reduce farmer's net income, but may also affect the supply of food and feed as well as the economies of rural areas and even countries.

Breeding of high-yielding varieties, use of synthetic fertilizers, and improved irrigation have contributed to double the world food production within the last 40 yr to match the demands of an increasing human population. Diverse ecosystems have been replaced in many regions by agroecosystems vulnerable to pest attack, especially when crops are grown in large-scale monocultures. Owing to limitations in resources such as yield potential of crops and availability of arable land and water, sustainability of production at elevated levels is only possible with adequate pest control. Loss data are the prerequisite for an economic management of pests and for evaluating the efficacy of the present crop protection practices. Based on this data, strategies for the use of limited resources may be developed to optimize productivity.^[2,3] The assessment of crop losses despite actual crop protection strategies is needed for demonstrating where action is needed and for decision-making.^[4] Estimates of actual losses in crop production worldwide have been published by Cramer^[5] and Oerke et al.^[6] Because crop production technology and especially crop protection methods available are changing, loss estimates for six major food and cash crops have been updated for the period 2001–2003.

METHODOLOGY

Data on area harvested, yield per unit of area, and total production for the six crops are obtained from

the Food and Agriculture Organization.^[7] The three-year average for the period 2001–2003 was used for further calculations. Crop losses owing to weeds, animal pests (arthropods, nematodes, mammals, slugs and snails, and birds), and pathogens (viruses, bacteria, fungi, and chromista) have been estimated from literature data.^[6] Literature inquiries in 1998, 2000, and 2004 have been used to update the information.

Two loss rates have been differentiated; the loss potential of pests includes the losses without physical, biological, or chemical crop protection using the similar intensity of crop production (fertilization, irrigation, cultivars, etc.) in a *no-loss scenario*. Actual losses comprise the crop losses despite the actual control practices. The calculation of total loss rates has been described earlier.^[6] For all crops, losses were calculated for 19 regions specified according to the intensity of crop production and production conditions: North Africa, West Africa, East Africa, Southern Africa; North America, Central America, northern part of South America, southern part of South America; Near East, South Asia, Southeast Asia, East Asia, and Asian states of Commonwealth of Independent States (CIS); West Europe, Southern Europe, East Europe, Southeast Europe, and European part of CIS; and Oceania.

LOSS POTENTIAL AND ACTUAL LOSSES OWING TO PESTS IN SIX MAJOR CROPS

Wheat

Weeds are the most important pests in wheat production worldwide. The incidence and impact of pathogens increase with the intensity of crop productivity (attainable yield). Arthropods, nematodes, rodents, birds, or snails cause significant losses only in some regions. Estimates of the loss potential of pathogens, animal pests, and weeds in wheat totalled 18%, 9%, and 23%, respectively (Table 1). Crop protection reduced the overall loss potential of 50% to actual losses of about 28%: 13% to pathogens, 8% to animal pests, and 8% to weeds. Total actual losses varied considerably, with 14% in Northwest Europe and >35% in the northern part of South America, Central Africa, Southeast Asia, and CIS (Fig. 1).

Table 1 Estimated loss potential of weeds, animal pests (arthropods, nematodes, rodents, birds, slugs, and snails) and pathogens (fungi, bacteria, and viruses), and actual losses owing to pest groups in six major crops worldwide, in 2001–2003

| Crop | Attainable production (M t) | Crop losses (%) ^a owing to | | | | | | | |
|----------|-----------------------------|---------------------------------------|----------------|-----------------|----------------|-----------------|-----------------|-----------------|-----------------|
| | | Weeds | | Animal pests | | Pathogens | | Total | |
| | | Potential | Actual | Potential | Actual | Potential | Actual | Potential | Actual |
| Wheat | 785 | 23 (18–29) | 7.7 (3–13) | 8.7 (7–10) | 7.9 (5–10) | 18.1 (15–24) | 12.6 (7–16) | 49.8 (44–54) | 28.2 (14–40) |
| Rice | 933.1 | 37.1 (34–47) | 10.2 (6–16) | 24.7 (13–26) | 15.1 (7–18) | 15.2 (10–17) | 12.2 (8–18) | 77 (64–80) | 37.4 (22–51) |
| Maize | 890.8 | 40.3 (37–44) | 10.5 (5–19) | 15.9 (12–19) | 9.6 (6–19) | 12.3 (9–18) | 11.2 (6–20) | 68.5 (58–75) | 31.2 (18–58) |
| Potatoes | 517.7 | 30.2 (29–33) | 8.3 (4–14) | 15.3 (14–20) | 10.9 (7–13) | 29.4 (28–33) | 21.1 (12–33) | 74.9 (73–80) | 40.3 (24–59) |
| Soybeans | 244.8 | 37 (35–40) | 7.5 (5–16) | 10.7 (4–16) | 8.8 (3–16) | 12.4 (7–18) | 10 (3–17) | 60 (49–69) | 26.3 (11–49) |
| Cotton | 78.5 ^b | 35.9 (35–39) | 8.6 (3–13) | 36.8 (35–41) | 12.3 (5–22) | 9.2 (7–10) | 7.9 (5–13) | 82 (76–85) | 28.8 (12–48) |

^aFigures in brackets indicate range, variation among 19 regions.

^bSeedcotton.

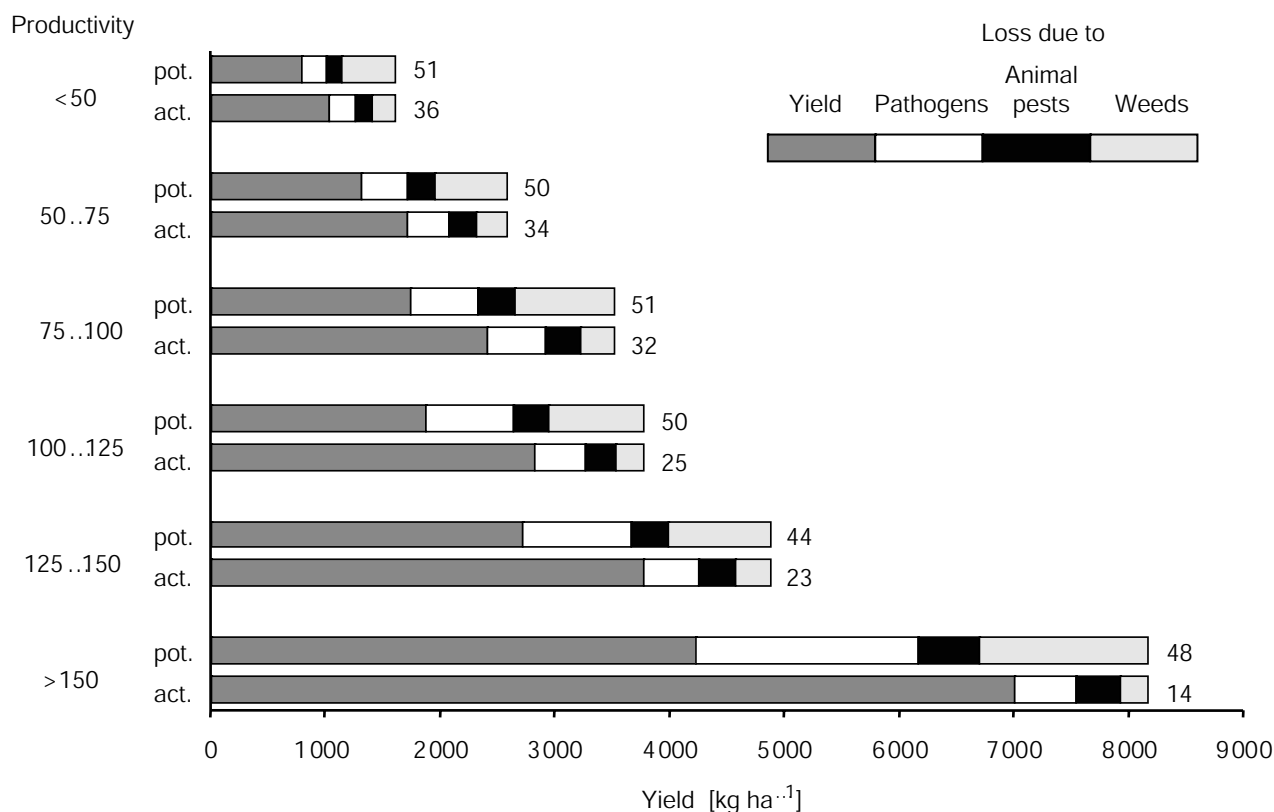


Fig. 1 Effect of intensity of wheat production on loss potential of and actual losses owing to pests in 2001–2003. Regions were grouped according to the actual yield level (as percentage of worldwide average): <50% (northern part of South America, Southeast Asia); 50–75% (West Africa, East Africa, Near East, Asian part of CIS, and Oceania); 75–100% (North Africa, South Africa, North America, southern part of South America, South Asia, South Europe, and European part of CIS); 100–125% (Southeast Europe); 125–150% (East Asia and East Europe); and >150% (Central America and West Europe). Figures on the right side of bars indicate total loss rates.

Rice

In rice production weeds, animal pests, and pathogens are regularly of economic importance; the estimates for the loss potentials averaged 37%, 25%, and 15%, respectively, worldwide (Table 1). Regional differences resulted from climatic conditions, cropping systems, and intensity of production, and the total loss potential of pests accounted for 64–80% of attainable yields. The variation in total actual loss rates—22% in Oceania and up to 51% in Central Africa—was considerably higher indicating significant differences in the efficacy of pest management. Weed control—mechanical or chemical—was effective in all regions, whereas the control of animal pests and diseases, which largely relies on pesticide use, showed great variation. Actual crop protection safeguarded about 40% of attainable rice production from being lost to pests. Nevertheless, actual losses at 37% of potential production remained high.

Maize

Worldwide maize production is threatened by the competition from weeds, which are the most important pest group (Table 1). With 40%, the loss potential was estimated to be higher than the sum of the loss potentials of animal pests and pathogens. Despite variation in weed species, the regional differences in loss potential were smaller than for animal pests and pathogens. For these pest groups, climatic conditions and geographical distribution of pests restrict the importance to some hot spots. Actual losses to weeds worldwide averaged 10% indicating low competitiveness of young maize seedlings as well as control problems in maize rotations where some species have become key pests. Actual losses to animal pests and pathogens, averaging 10% and 11%, respectively, showed greater variation than loss potentials. Worldwide, about 37% of attainable maize production was protected from being lost to pests; the percentage varied from 18% to 45% in South Europe and the U.S.A.

Potatoes

As vegetative propagation predominates in potato production, all pest groups are of high economic importance (Table 1). Loss estimates for pathogens, animal pests, and weeds worldwide totalled 21%, 11%, and 8%, respectively. Without crop protection, about 75% of attainable production would be lost to pests. Major fungal pathogens, viruses, and animal pests are widely distributed resulting in a low variation of total loss rates among regions. Actual total losses are estimated to vary from 24% in Northwest Europe to >55% in Central Africa. Manual, mechanical, and chemical pest

management protected about 35% of attainable potato production from destruction. The share reached only 21% in Central Africa, where pest control is largely restricted to the control of weeds, and amounted to >50% in North America and West Europe, where intensive crop protection allows high productivity. As the control of potato late blight, some viruses, and nematodes is still problematic, actual losses accounted for 40% of attainable production.

Soybeans

Weeds are the predominant pests in soybean production. About 37% of attainable production is threatened by weed competition worldwide compared to 12% and 11% by pathogens and animal pests, respectively (Table 1). Regional variation of loss rates for weeds was low, whereas variation in losses to pathogens and animal pests were estimated to be high because of the regionally restricted incidence of key pathogens and nematodes. As soybean rust has been invasive in South America since 2001—it has been confirmed also for the U.S.A. in November 2004—the impact of this destructive pathogen has dramatically increased in global soybean production within a short period of time. Worldwide, actual losses to pathogens and animal pests were estimated to be only slightly lower than the loss potentials, as crop protection in soybean has concentrated on weed control. Mechanical and chemical control reduced the loss potential of weeds by 80% to an average of 7%, varying from 5% in South Europe to 16% in Central Africa. Pest control protected almost 34% of attainable soybean production from destruction and increased worldwide production to 74% of the potential.

Cotton

Cotton production is threatened especially by insect attack and by weed competition during early stages of development. Pathogens may be harmful in some areas and during certain years, but are generally considered to be of minor importance (Fig. 2). The worldwide estimates for the loss potentials of animal pests and weeds averaged 37% and 36%, respectively (Table 1). Pathogens added about 9% to a total loss potential of 82%. The variation among regions was small indicating that successful cotton production without crop protection is not feasible. Actual losses to pathogens, animal pests, and weeds showed greater regional variability and totalled worldwide 8%, 12%, and 9%, respectively. The share of cotton production protected by actual pest control practices was calculated at 53%. The contribution of crop protection in cotton production varied from 37% in Central Africa to 68% in Australia.

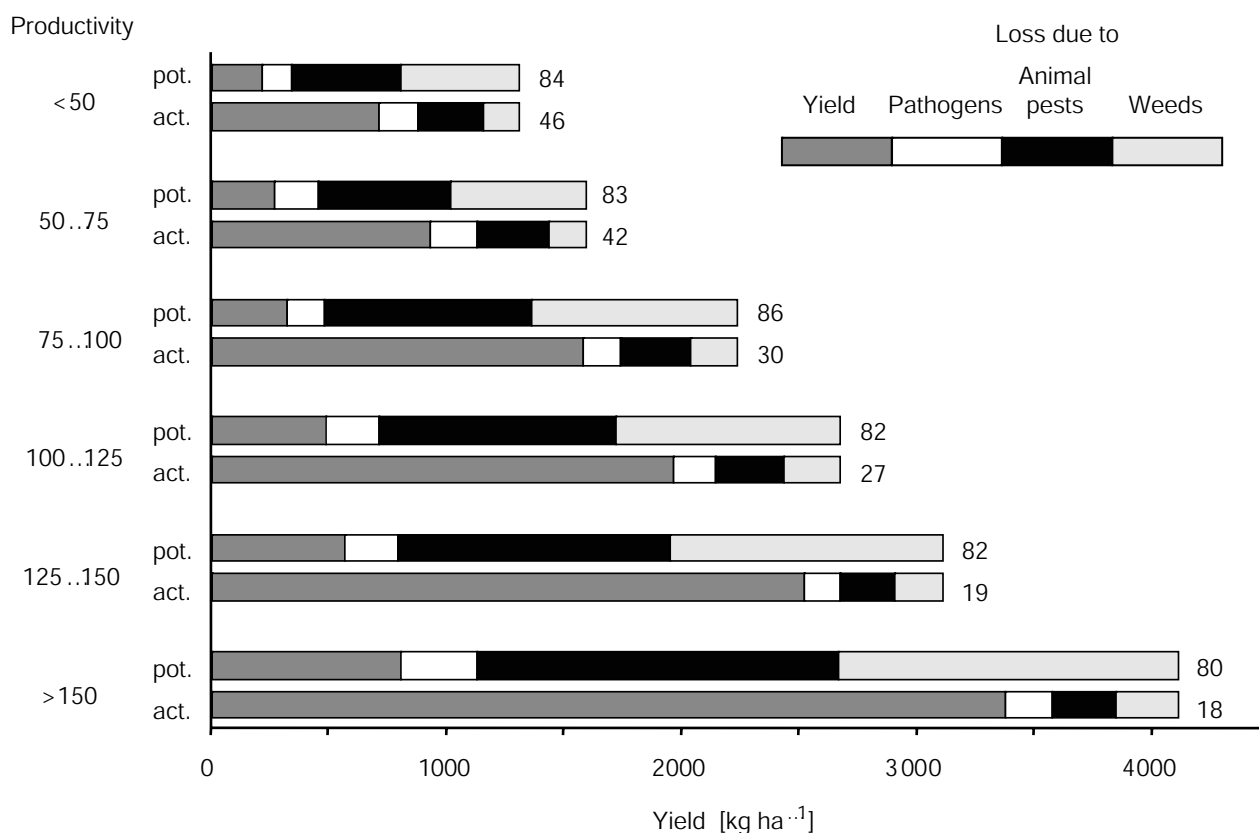


Fig. 2 Effect of intensity of cotton production on loss potential of and actual losses owing to pests in 2001–2003. Regions were grouped according to the actual yield level (as percentage of worldwide average): <50% (East Africa, South Africa, and Southeast Asia); 50–75% (West Africa, South Asia, Southeast and Europe); 75–100% (northern part of South America); 100–125% (North America, southern part of South America, and Asian part of CIS); 125–150% (North Africa and Central America); and >150% (Near East, East Asia, South Europe, and Oceania). Figures on the right side of bars indicate total loss rates.

REGIONAL VARIATION IN PRODUCTIVITY AND CROP LOSSES

The loss potential of pests increases with the intensity of production (attainable yield), especially for pathogens, favored by ample biomass production, not only absolute losses (in kg/ha) rise but also loss rates (% of attainable yield), whereas the impact of weeds tends to decrease under intensive farming conditions (Fig. 1). To safeguard productivity in the presence of high competition, growers bestow great care on pest control, effectively reducing losses to an economically acceptable level. Climatic conditions promoting the incidence, survival, and propagation of pests in the tropics and subtropics, differences in the knowledge and training of farmers, and the access to effective pesticides, intensify the differences between cropping regions. However, regional differences—in absolute yields and losses—are less pronounced in cash crops, e.g., cotton (Fig. 2), than in crops like wheat grown for food and feed. This is especially true for developing countries

where food production often lacks the support the production of cash crops is receiving.

CONCLUSIONS

Crop losses to weeds, animal pests, and pathogens reduce crop production and limit the availability of food and feed especially in developing countries. The overall loss potential is especially high in crops grown under high productivity conditions as well as in the tropics and subtropics. The intensity of crop protection depends on the importance of the pests or its perception by growers and on the availability of crop protection tools. As the availability of control measures greatly varies among regions, actual losses despite pest control measures differ to a higher extent than site-specific loss potentials. The intensity and efficacy of crop protection has increased recently, especially in Asia and Latin America where the use of pesticides increased from 1993 to 1998 by 5.4% annually, well

above the global average of 4.4%.^[8] New compounds highly effective against pests formerly less controllable, the use of genetically modified crops (GMOs) especially in North America and Asia, where China is the country with the highest growth in land cropped with GMOs,^[9] and a better training of farmers have contributed to an improvement in pest control. However, the situation is still unfavorable in sub-Saharan Africa.

As the level of crop losses economically acceptable in most field crops is well above zero, in Integrated Pest Management, the use of pesticides is based on the use of economic threshold levels.^[10] Some losses cannot be avoided because of the lack of control options; others have to be accepted because of ecological hazards associated with a potential use. In many cases, however, a higher pesticide use to produce extra yield from preventing crop losses is economically not justified because other environmental factors than pests, especially water availability, are yield limiting. In many regions a reduction of loss rates seems to be desirable to improve the food supply of peoples; however, growers apply pesticides for their economic benefit, which is modest at low intensity of crop production.

In many regions the crop productivity has to be increased to meet a growing demand. As increased yield potentials are often associated with higher vulnerability of crops to pest attack, the increased threat of higher crop losses to pests has to be counteracted by improved crop protection by whatever method is required—biologically, mechanically, chemically, or training of growers and advisors in Integrated Pest Management.

REFERENCES

1. Zadoks, J.C.; Schein, R.D. *Epidemiology and Plant Disease Management*; Oxford University Press: Oxford, UK, 1979.
2. Cooke, B.M. Disease assessment and yield loss. In *The Epidemiology of Plant Diseases*; Jones, D.G., Ed.; Kluwer Publishers: Dordrecht, Netherlands, 1998; 42–72.
3. Nutter, F.W., Jr.; Teng, P.S.; Royer, M.H. Terms and concepts for yield, crop loss, and disease thresholds. *Plant Dis.* **1993**, *77*, 211–215.
4. Smith, I.M.; Chiarappa, L.; van der Graaff, N.A. World crop losses: an overview. In *Plant Diseases: Infection, Damage and Loss*; Wood, R.K.S., Jellis, G.J., Eds.; Blackwell Scientific Publications: Oxford, UK, 1984; 213–223.
5. Cramer, H.H. Plant protection and world crop production. *Bayer Pflanzenschutz-Nachrichten* **1967**, *20*, 1–524.
6. Oerke, E.-C.; Dehne, H.-W.; Schönbeck, F.; Weber, A. *Crop Production and Crop Protection—Estimated Losses in Major Food and Cash Crops*; Elsevier Science: Amsterdam, Netherlands, 1994.
7. <http://apps1.fao.org/servlet/> (accessed March 2004).
8. Yudelman, M.; Ratta, A.; Nygaard, D. Pest management and food production. Looking to the future. In *Food, Agriculture, and the Environment*; International Food Policy Research Institute: Washington, DC, 1998, Discussion Paper 25.
9. McLaren, J.S. The importance of genomics to the future of crop production. *Pest Manag. Sci.* **2000**, *56*, 573–579.
10. Oerke, E.-C.; Dehne, H.-W. Safeguarding production—losses in major crops and the role of crop protection. *Crop Protection* **2004**, *23*, 275–285.

Crop Residues and Pest Problems

Alexandra Wilson-Rummenie

Bruce Radford

Department of Natural Resources, Bileola Research Station,
Bileola, Australia

INTRODUCTION

The retention of crop residues modifies the soil micro-environment, which changes the population density and species composition of the soil macrofauna. The species composition of soil-dwelling pests also changes but the total number of pests and the pest problem do not necessarily increase. Residue retention encourages the proliferation of predatory insects. Some insect species feeding on crop residues switch to crop seedlings (e.g., wingless cockroaches) but others do not (e.g., false wireworms, particularly when ample residues are still present). The use of germinating grain as bait to assess a pest problem is simple and detects all insect pests. Control measures include residue management, crop rotation, weed control during the fallow, the use of press wheel pressure at sowing, and the application of insecticides. Methods of insecticide application vary with the target species and include seed dressing, seed soaking, water injection, in-furrow spraying or granular application, and distribution of insecticidal grain bait on the soil surface.

NEED FOR CROP RESIDUES

Retention of protective crop residues on the soil surface greatly reduces the problem of soil erosion, which is a serious threat to sustainable crop production. For cultivated agriculture, adequate cover reduces soil erosion more than any other factor in tillage management.^[1] Critical cover levels required to reduce soil erosion to minimum levels have been quoted as: 30%,^[1] 40%,^[2] 50–70%,^[3] or 2–4 tn/ha.^[4]

Management practices that maximize levels of cover include no tillage and controlled traffic. Each pass of a tillage implement buries residues. A one-way disk plough reduces cover by 50–65%, a chisel plough fitted with sweeps by 12–50%, a scarifier by 20–30%, blade and sweep ploughs by 10–20%, and a rod weeder by 10–15%.^[5] Management to control erosion should take precedence over practices designed specifically to control soil-dwelling pests.

ROLE OF CROP RESIDUES IN MODIFYING THE SOIL MICROENVIRONMENT

Crop residues (commonly known as trash or stubble) modify the soil microenvironment in a number of ways. Residues on the soil surface may increase infiltration,^[5] retard evaporation,^[6] and moderate soil temperatures.^[7] Residues provide additional substrate allowing a higher population density of soil-inhabiting detritivores, which in turn provide additional prey for soil-inhabiting predators. Residues also create protective habitat for predatory insects such as carabids and rove beetles.^[8]

These modified soil physical conditions in combination with no tillage have doubled the population density of soil macrofauna compared with no surface cover and traditional tillage during the fallow.^[5] The greater numbers of macrofauna further modify the physical and chemical properties of the soil.^[9]

Earthworms have a particularly beneficial effect on soil structure,^[10] and earthworm activity is greater under conservation tillage practices, such as reduced and zero tillage, than conventional tillage systems.^[11] Their burrows increase macroporosity and consequently water infiltration and aeration.^[12]

EFFECT OF CROP RESIDUES ON SOIL PEST POPULATIONS

The retention of crop residues favors those insect pests, which feed on both residues and seedling field crops.^[13] Larvae of false wireworms (*Pterohelaeus*, *Gonocephalum* and *Cestrinus* spp.) feed on plant residues but will switch to germinating seeds and emerging seedlings in the absence of abundant organic matter. Several soil-inhabiting larvae, including wireworms and scarabs, are attracted to sources of carbon dioxide. This is why they are found on decomposing crop residues and on germinating seeds.^[14]

Wingless cockroaches (*Calolampra* spp.), scarab grubs, black field earwigs (*Nala lividipes* Dufour), and field crickets all use litter and crop residues as

a food source but switch to green grasses, broad-leaved annuals, and emerged crop seedlings when these are available. Adult false wireworms continue to feed on stubble if it is retained on the soil surface at sowing (no tillage) rather than switch to feeding on the emerged seedlings. A given population density of false wireworm beetles causes fewer seedling losses in crops direct-drilled through stubble than in crops sown into bare soil.^[14] Wingless cockroaches feeding on stubble, however, will switch to crop seedlings even with direct drilling. Black field earwigs are a major pest that do not rely on stubble to maintain a high population and a pest even with stubble burning.^[15]

Conservation tillage generally does not cause an increase in total soil pest population.^[16-17] There is a change only in the species composition of the pests.

Recent research has shown that a wheat-cotton rotation saves one to three applications of endosulfan for heliothis control; a likely explanation is that the wheat stubble acts as a physical barrier preventing heliothis moths from finding the cotton plants.^[18] Once the cotton emerges above the standing wheat stubble, there is no further advantage in insect control. This suggests that leaving taller wheat stubble will prolong nonchemical control. Total predator numbers were also found to be 20% higher in the cotton grown in wheat stubble.

ASSESSMENT OF SOIL PEST POPULATIONS

Soil insect populations can be assessed by spade sampling, which is laborious and time-consuming, or by using germinating grain as bait, which is simple and accurate and detects all soil insect pests.

The procedure for the germinating grain bait technique^[19] is:

1. Soak crop seed (free of insecticides) in water for at least 2 hr to initiate germination.
2. Bury the seed at shallow depth in the field and cover lightly with 1 cm of soil.
3. Bury one small handful of seed on the corners of a 5 × 5 m area (marked with pegs).
4. Repeat at five widely spaced sites in each 100 ha.
5. Dig up the seedlings a day after emergence.
6. Place the plants and soil in a tray and count the insects.

For summer crops, control measures are warranted when there are one or more insects per bait, or five earwigs. For winter crops, control is warranted when there are two or more insects per bait, or ten earwigs. Ant control is warranted when infestations occur on 50% of baits.

CONTROL MEASURES

Insecticides applied to control soil insect pests also adversely affect soil-inhabiting predators and decomposers.^[14] Control measures, which are less damaging to the environment are needed.

Burning of crop residues or tillage should not be used to control soil insect pests because these practices reduce ground cover. Soil erosion and soil structural decline are more serious long-term constraints to sustainable cropping than transient insect pest problems. Cultivation has been recommended to control crop pests with a soil-inhabiting stage, including heliothis pupae,^[20] but stalk-pulling in cotton stubble is sufficient to kill heliothis pupae.

Crop rotation using botanically diverse crop species limits the population increase of soil insect pests. Wheat in rotations leads to lower average false wireworm populations and sorghum leads to higher populations because false wireworms and most other soil insect pests prefer sorghum residues.^[14]

Weeds and volunteer crop plants during the fallow encourage the proliferation of soil insect pests. Effective weed control during the fallow helps reduce pest numbers but weed control by tillage also reduces residue levels and hence ground cover.

The use of press wheels at sowing has effectively controlled soil insect pests.^[21] Increases in press wheel pressure improve the level of control but can only be used in crops capable of emerging through compacted soil.

When insecticides are needed to control soil pests of seeds and seedlings, they can be applied as a seed dressing by soaking the seed (if the insecticide is not phytotoxic), by water injection, or by in-furrow spray or granule application. Soil treatments are generally more effective than seed treatments, and insecticides with systemic action are best.

Surface-feeding insects can be controlled with insecticidal grain bait spread on the soil surface. A common mix is 100 ml chlorpyrifos (500 g/L) EC and 125 ml crop or vegetable oil in 2.5 kg of cracked wheat, sorghum, or standard pellets.^[19] Once mixed, the bait is applied at 2.5 kg/ha with a fertilizer spreader or through fertilizer tubes. Even distribution is needed for effective control.

REFERENCES

1. Freebairn, D.M.; Loch, R.J.; Cogle, A.L. Tillage methods and soil and water conservation in Australia. *Soil Tillage Res.* **1993**, *27*, 303-325.
2. Gardener, C.J.; McIvor, J.G.; Williams, J. Dry tropical rangelands: solving one problem and creating another. *Proc. Ecol. Soc. Aust.* **1990**, *16*, 279-286.

3. Lang, R.D.; McCaffrey, L.A.H. Ground cover—Its effects on soil loss from grazed runoff plots, Gunnedah. *J. Soil Conserv. Serv. N.S.W.* **1984**, *40*, 56–61.
4. Lal, R. Soil erosion on alfisols in western Nigeria. *Geoderma* **1976**, *16*, 377–378.
5. Radford, B.J.; Key, A.J.; Robertson, L.N.; Thomas, G.A. Conservation tillage increases soil water storage, soil animal populations, grain yield, and response to fertiliser in the semi-arid subtropics. *Aust. J. Exp. Agric.* **1995**, *35*, 223–232.
6. Bond, J.J.; Willis, W.O. Soil water evaporation: surface residue rate and placement effects. *Soil Sci. Soc. Am. Proc.* **1969**, *33*, 445–448.
7. McCalla, T.M.; Army, T.J. Stubble mulch farming. *Adv. Agron.* **1961**, *13*, 126–196.
8. House, G.J.; Brust, G.E. Ecology of low-input, no-tillage agroecosystems. *Agric. Ecosyst. Environ.* **1989**, *27*, 331–345.
9. McGarry, D.; Bridge, B.J.; Radford, B.J. Contrasting soil physical properties after zero and traditional tillage of an alluvial soil in the semi-arid subtropics. *Soil Tillage Res.* **2000**, *53*, 105–115.
10. Lal, R. Effects of macrofauna on soil properties in tropical ecosystems. *Agric. Ecosyst. Environ.* **1988**, *24*, 101–116.
11. Lal, R. No-tillage effects on soil properties under different crops in western Nigeria. *Agric. Ecosyst. Environ.* **1976**, *48*, 762–768.
12. Lee, K.E.; Foster, R.C. Soil fauna and soil structure. *Aust. J. Soil Res.* **1991**, *29*, 745–777.
13. Allsopp, P.G. False wireworms in southern and central Queensland. *Qld. Agric. J.* **1979**, *105*, 276–278.
14. Wildermuth, G.B.; Thompson, J.P.; Robertson, L.N. Biological change: diseases, insects and beneficial organisms. In *Sustainable Crop Production in the Subtropics*; Clarke, A.L., Wylie, P.B., Eds.; Information Series QI97035; Queensland Department of Primary Industries: Brisbane, 1997; 122–124.
15. Robertson, L.N. Effects of crop stubble on soil-insect populations in Queensland. In *Proceedings of a Soil Invertebrate Workshop*; Robertson, L.N., Allsopp, P.G., Eds.; Queensland Department of Primary Industries Conference and Workshop Series QC89004, 1989; 26–28.
16. Robertson, L.N.; Kettle, B.A.; Simpson, G.B. The influence of tillage practices on soil macrofauna in a semi-arid agroecosystem in northeastern Australia. *Agric. Ecosyst. Environ.* **1994**, *48*, 149–156.
17. Wilson-Rummenie, A.C.; Radford, B.J.; Robertson, L.N.; Simpson, G.B.; Bell, K.L. Reduced tillage increases population density of soil macrofauna in a semiarid environment in central Queensland. *Environ. Entomol.* **1999**, *28*, 163–172.
18. Waters, D.; Drysdale, R.; Kimber, K. Planting cotton into wheat stubble. *Aust. Grain* **1999**, *9*, iv–vi.
19. Sequeira, R.; Muller, P. Control of soil insects. In *Crop Management Notes Central Queensland 1999*; Harris, G., Ed.; Information Series QI99001; Queensland Department of Primary Industries: Brisbane, 1999; 42–46.
20. Fitt, G.P. The ecology of heliothis species in relation to agroecosystems. *Annu. Rev. Entomol.* **1989**, *34*, 17–52.
21. Radford, B.J.; Allsopp, P.J. Use of insecticides and a press wheel to control soil insects affecting sorghum and sunflower establishment in southern Queensland. *J. Aust. Entomol. Soc.* **1987**, *26*, 161–167.

Crop Rotations for Weed Control

Paul Mugge

Sutherland, Iowa, U.S.A.

INTRODUCTION

Crop rotation is the temporal diversification practice of growing a sequence of plant species on the same land.^[1,2] Crop rotation can be a valuable part of an integrated weed control strategy. Weed populations utilize specific ecological niches that are similar to the crops in which they proliferate, or they take advantage of conditions associated with that crop. Sequences of crops, particularly those with different life cycles and management requirements, cause unstable environments for weeds and deny them the opportunity to take advantage of a system for which they are particularly adapted.^[3] Summer annual weeds, for example, predominate in a corn/soybean rotation because both crops and weeds are summer annuals.^[1,2,4] Because crop rotation provides continuous habitat modification, no one weed can dominate, and evenness of a wider diversity of species is favored.^[5]

ROTATION EFFECTS

Weed population ecology is affected by three interacting groups of extrinsic factors:^[6]

1. management factors
2. weather
3. interactions with other organisms (insects, pathogens, other plants, herbivores)

Together, these interacting biotic and abiotic factors produce shifts in weed populations over time, and it is difficult to separate the effects of the rotation itself from the effects of the management factors concomitant with the various crops. Doucet et al.^[7] attempted to separate the effects of crop rotation from other weed management factors and determined that for a corn/soybean/winter wheat rotation, weed management accounted for 37.9% of the variation in weed density, while crop rotation accounted for only 5.5%. Conversely, in the few studies examining the effects of rotated crops apart from those of rotated herbicides, crop rotation usually led to reduced weed populations. This is especially true when the rotation includes a small grain.^[2,8,9] Cardina, Herms, and Doohan^[8] found that crop rotation was a more important determinant of soil seed density than

was tillage, but Dorado, Del Monte, and Lopez-Fando,^[10] working with barley/vetch, barley/sunflower, and barley monoculture in central Spain, found that 6 of 36 weed species significantly differed as a result of rotation, while 11 of 36 responded to tillage. Although Dorado, Del Monte, and Lopez-Fando,^[10] found different dominant weeds in the different rotations, fall-seeded barley/vetch produced more abundant and diverse weed populations than either barley/sunflower or barley monoculture. The authors attributed the response to greater competitiveness of weeds with vetch, especially for light, and possibly greater N availability because of the legume. Other studies have shown increases in weed parameters in rotations that include short-term forages. Liebman et al.^[4] studied potato following either oat or berseem clover. Weed biomass in the oat or clover phase, soil seed density between the phases, and weed density and biomass in the potato phase all tended to be higher following berseem clover than following oat, although there were exceptions. Stevenson et al.^[11] compared a barley/forage (timothy/red clover) rotation with barley monoculture in Quebec, Canada. Although residual weed dry weight was higher in the barley/forage rotation, probably as a result of higher herbicide usage in the barley monoculture, barley seed yields were also higher when a forage was included.

ECOLOGICAL MECHANISMS

Competition

Competition for resources is probably the most common mechanism regulating the relationships among various plants. All plants, crops and weeds, have specific biological requirements necessary to survive and thrive. Some plants use a very narrow niche and others may be able to utilize a broad range of resources. When species interact, to the extent that they cannot divide or share the niche, they will compete for resources such as light, water, nutrients, space, etc. The species able to more efficiently utilize a resource, or survive at a lower level of that resource, will have an advantage. Crop plants vary in their ability to out-compete various weed plants because they vary in their ability to use resources relative to weeds.^[12] Different varieties of the same crop also differ in competitiveness. Those that are taller, for

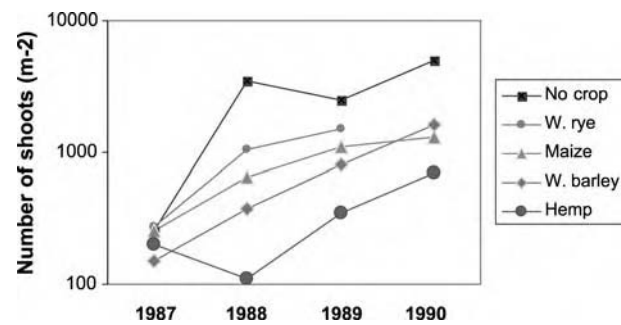


Fig. 1 The after-effect of different crop treatments in 1987 on the density of primary shoots of *C. esculentus* in maize crop in subsequent years. (From Ref.^[13].)

example, or have a higher leaf area index, are better able to shade weeds below their canopy.

Lotz et al.^[13] conducted a fascinating study in the Netherlands. They raised four different crops in 1987 (silage corn, hemp, winter barley, and silage winter rye) before corn in 1988–1990, to identify differences in the crops' ability to control yellow nutsedge (*Cyperus esculentus*). As can be seen in Fig. 1, yellow nutsedge was dramatically reduced in corn following hemp relative to the other preceding crops. In the greenhouse, Lotz simulated the intensity of light measured under the various crops, and produced a similar effect on yellow nutsedge, indicating that competition for light was likely responsible for the weed reduction. Clay and Aguilar^[3] showed that grass and broadleaf weeds were greatly suppressed in alfalfa in the year following establishment. In corn following the alfalfa under low-input chemical weed control, grass weeds were reduced by about 90% on average in an alfalfa/corn rotation vs. corn/corn. Similarly, broadleaf weeds averaged 70% lower. Liebman Mohler, and Staver^[12] listed several other examples of weed declines following perennial forages, but cautioned that other weeds such as dandelion (*Taraxacum officinale*) and stinkweed (*Thlaspi arvense*) may show increases.

Allelopathy

Some plants produce natural toxins, called allelochemicals, which inhibit the germination or growth of other plants. This phenomenon, called allelopathy, can assist in weed control.^[1,12] When soybeans and sunflower were planted no-till into killed green rye, lambsquarter (*Chenopodium album*) growth was reduced 99%, pigweed (*Amaranthus retroflexus*) 96%, and common ragweed (*Ambrosia artemisiifolia*) 92%, compared with tilled plots with no mulch.^[14] Davis and Liebman^[15] investigated the interference of wild mustard (*Brassica kaber*) with sweet corn in central Maine, United States.

In comparing sources of N, they found that organic sources reduced negative weed effects on sweet corn yield compared to inorganic sources either in split application or single early treatment. Because wild mustard seedling emergence and sweet corn yield loss were very negatively correlated with the amount of red clover biomass incorporated into the soil, they speculated that allelochemicals in the decomposing red clover inhibited germination or emergence of wild mustard, although it was not directly shown. Schreiber^[9] found significantly reduced giant foxtail (*Setaria faberi*) seed and plant density in soybean/corn relative to corn/corn and less still in soybean/wheat/corn. Although significant giant foxtail reductions were seen in a chisel plow system, dramatic results are seen in the no-till system under minimum weed management levels, which Schreiber attributes to the allelopathic effect of the unincorporated wheat straw (Figs. 2A–2C).

Soil Quality Effects

Rotating crops produces changes in soil physical, chemical, and biological characteristics, which in turn change the habitat available to weed seeds, and eventually the demographics of weed populations. Tillage and residue cover affect seed mortality, for example, by affecting burial depth, predation, climatic exposure, etc.

Theoretically, crop management schemes that provide N closer to the time it is used by the crop would provide weed control benefits relative to a system in which large amounts of N were available early in the year. Weeds, with seeds that are typically small compared to those of the crop, mitigate their initial disadvantage by high early nutrient uptake and growth, and should be at a disadvantage relative to the crop in an environment in which nutrients are limited early in the season. Although some research has substantiated those benefits, any effect is most often quite small. Stevenson et al.^[11] found no difference between manure and mineral fertilizer on total weed biomass, and Davis and Liebman^[15] found only limited support for the delayed N effect when comparing organic nutrient sources with either a split application or large early application of NH_4NO_3 .

Cultural Effects

Rotation of cultural practices is incumbent upon rotations of crops. Chancellor^[16] monitored changes in weed flora in England for 20 years of arable cropping after being plowed out of permanent grass. He found that season of crop planting was one of the most important determinants of weed flora. Spring-germinating weeds predominate in spring-seeded crops and vice versa. Fall-germinating weeds are killed by preplant

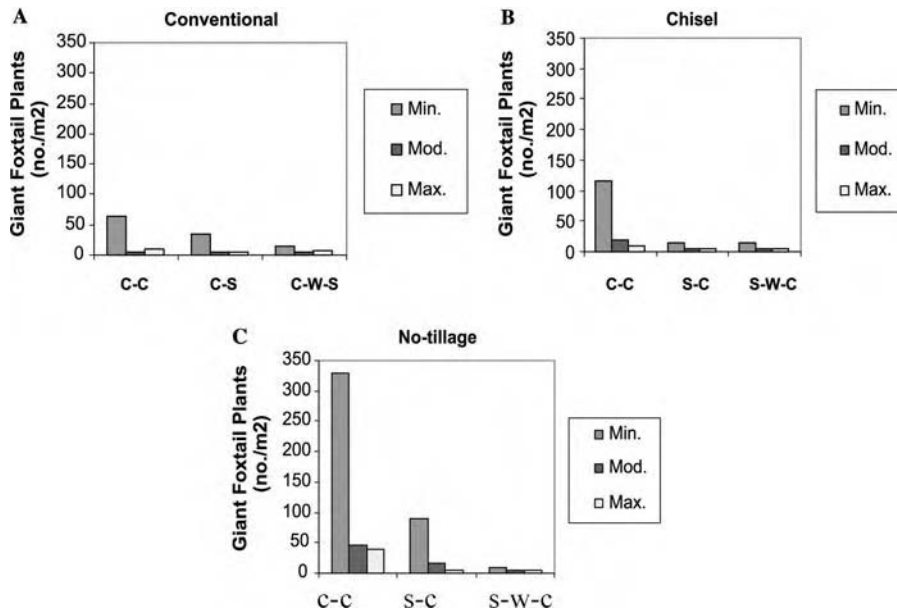


Fig. 2 Giant foxtail stands in untreated areas as influenced by crop rotation and weed management levels under conventional (A), chisel (2B), and no-tillage (C) conditions in 1986. (From Ref.^[9].)

tillage before spring-seeded crops and spring-germinating weeds emerge into the established canopy of fall-seeded crops.^[6,10] Crop rotations allow for rotations of tillage and herbicides, which very effectively alter weed population dynamics.^[7,8]

Cumulative Effects

Ultimately, weeds are presented with a suite of crops and management practices within a crop rotation. In work with continuous corn, corn/soybean, and corn/tomato/soybean rotations, Manley, Wilson, and Hines^[17] attributed control of four small seeded broad-leaf weeds to the interaction between crop rotations and herbicide programs. Likewise, Ball^[18] showed that a combination of rotated crops and herbicides produced shifts in the weed seedbank. Downy brome (*Bromus tectorum*) is a serious weed in winter wheat on the Canadian prairies. Blackshaw compared continuous winter wheat with wheat rotated with either fallow or

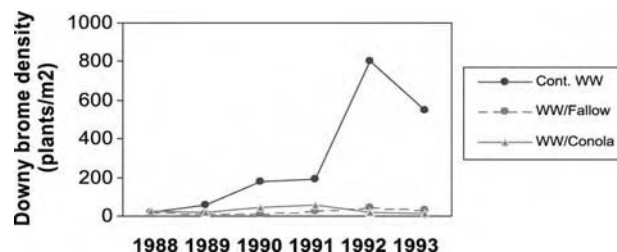


Fig. 3 Downy brome density in winter wheat grown continuously or in rotation with fallow or spring canola under conventional tillage. (From Ref.^[19])

spring canola.^[19] Results showed dramatic decreases of downy brome in both crop rotations compared to monoculture (Fig. 3).

CONCLUSION

Although crop rotations may increase evenness and lessen the chance of one weed becoming dominant, they probably cannot adequately limit total weed numbers, biomass, or seed production by themselves. Rotating crops provide an opportunity to rotate crop life cycle (fall-seeded and spring-seeded, annual and perennial, close-seeded and row-crop) as well as management (tillage, herbicides, nutrient sources, plant/harvest dates).

It is hoped that future research will develop new cropping systems and new insight into using them to take advantage of the synergies among the many interacting mechanisms. Near-term crop rotations provide an essential element of an integrated system of weed management necessary for more sustainable agroecosystems.

REFERENCES

1. Bullock, D.G. Crop rotation. *Crit. Rev. Plant Sci.* **1992**, *11* (4), 309–326.
2. Liebman, M.; Dyck, E. Crop rotation and intercropping strategies for weed management. *Ecol. Appl.* **1993**, *3* (1), 92–122.
3. Clay, S.A.; Aguilar, I. Weed seedbanks and corn growth following continuous corn or alfalfa. *Agron. J.* **1998**, *90*, 813–818.

4. Liebman, M.; Drummond, F.A.; Corson, S.; Zhang, J. Tillage and rotation crop effects on weed dynamics in potato production systems. *Agron. J.* **1996**, *88*, 18–26.
5. Legere, A.; Samson, N. Relative influence of crop rotation, tillage, and weed management on weed associations in spring barley cropping systems. *Weed Sci.* **1999**, *47*, 112–122.
6. Cousens, R.; Mortimer, M. *Dynamics of Weed Populations*; Cambridge University Press: Cambridge, UK, 1995.
7. Doucet, C.; Weaver, S.E.; Hamill, A.S.; Zhang, J. Separating the effects of crop rotation from weed management on weed density and diversity. *Weed Sci.* **1999**, *47*, 729–735.
8. Cardina, J.; Herms, C.P.; Doohan, D.J. Crop rotation and tillage system effects on weed seedbanks. *Weed Sci.* **2002**, *50*, 448–460.
9. Schreiber, M.M. Influence of tillage, crop rotation, and weed management on giant foxtail (*Setaria faberi*) population dynamics and corn yield. *Weed Sci.* **1992**, *40*, 645–653.
10. Dorado, J.; Del Monte, J.P.; Lopez-Fando, C. Weed seedbank response to crop rotation and tillage in semi-arid agroecosystems. *Weed Sci.* **1999**, *47*, 67–73.
11. Stevenson, F.C.; Legere, A.; Simard, R.R.; Angers, D.A.; Pageau, D.; Lafond, J. Manure, tillage, and crop rotation: effects on residual weed interference in spring barley cropping systems. *Agron. J.* **1998**, *90*, 496–504.
12. Liebman, M.; Mohler, C.L.; Staver, C.P. *Ecological Management of Agricultural Weeds*; Cambridge University Press: Cambridge, UK, 2001.
13. Lotz, L.A.P.; Groeneveld, R.M.W.; Habekotte, B.; Van Oene, H. Reduction of growth and reproduction of *Cyperus esculentus* by specific crops. *Weed Res.* **1991**, *31*, 153–160.
14. Worsham, A.D. Crop residues kill weeds. *Crops Soils Mag.* **1984**, *37* (2), 18–20.
15. Davis, A.S.; Liebman, M. Nitrogen source influences wild mustard growth and competitive effect on sweet corn. *Weed Sci.* **2001**, *49*, 558–566.
16. Chancellor, R.J. Changes in the weed flora of an arable field cultivated for 20 years. *J. Appl. Ecol.* **1985**, *22*, 491–501.
17. Manley, B.S.; Wilson, H.P.; Hines, T.E. Weed management and crop rotations influence populations of several broadleaf weeds. *Weed Sci.* **2001**, *49*, 106–122.
18. Ball, D.A. Weed seedbank response to tillage, herbicides, and crop rotation sequence. *Weed Sci.* **1992**, *40*, 654–659.
19. Blackshaw, R.E. Rotation affects downy brome (*Bromus tectorum*) in winter wheat (*Triticum aestivum*). *Weed Technol.* **1994**, *8* (4), 728–732.

Cross-Resistance to Pesticides

Bruce J. Cochran

Biology Department, University of South Florida, Tampa, Florida, U.S.A.

INTRODUCTION

Cross-resistance to pesticides occurs when a particular biological mechanism confers resistance to multiple pest control agents. In some cases, this is due to the structural similarity of the compound involved: For example, there are large numbers of synthetic pyrethroids that all have the same mode of toxicity, so that populations may become resistant to a large suite of these compounds as a result of selection resulting from the use of only one. In other cases, structurally distinct compounds (e.g., organophosphates and carbamates) may also have common physiological targets, so that prior use of one in a control program may in fact select for a mechanism that confers resistance to the other.

Cross-resistance should be distinguished from multiple-resistance, in which a population displays resistance to a variety of control agents, but the mechanisms conferring resistance to each are distinct. In addition, there are reports of negative cross-resistance, in which resistance to one control agent is accompanied by sensitivity to others. This is particularly prevalent with respect to herbicide resistance in plants in which, in contrast to pest insects, cross- and multiple-resistance are in fact desirable traits in crop plants.

MECHANISMS OF CROSS-RESISTANCE

Most major physiological mechanisms of pesticide resistance fall into one of two categories—target-site modification or changes in metabolic detoxification pathways. The existence of common target sites is the most common source of cross-resistance. Both DDT and pyrethroids act by binding to and disrupting the voltage-gated sodium channel in insects, thus leading to neurotoxicity. Similarly, organophosphates such as malathion and carbamates such as carbaryl both function by inhibiting acetylcholinesterase, again causing neurotoxicity. Finally, three compounds—dieldrin (a cyclodiene), toxaphene, and lindane—target the GABA-gated chloride channel, as do more recently developed abamectins.

In the case of metabolic mechanisms, the problem is much more complex. The enzymes involved—primarily glutathione S-transferases, esterases, and cytochromes *P450*—are encoded by large gene families (there are over 80 *P450*-like sequences in the genome of *Drosophila melanogaster*), so that it is difficult to determine which isoform is responsible for a given resistance mechanism, let alone for cross-resistance. In those cases where metabolic cross-resistance has been detected, the level of resistance is typically low. Finally, metabolic processes typically confer cross-resistance to a small number of insecticides within a given class.

It is important to recognize that the co-occurrence of resistance to multiple classes of pesticides cannot be accepted as evidence for true cross-resistance. In one strain of *Drosophila simulans*, resistance to malathion and carbaryl, both acetylcholinesterase inhibitors, is in fact due to clearly distinct mechanisms. Malathion resistance is polygenic in nature, and the presence of an insensitive form of acetylcholinesterase makes a major contribution. On the other hand, carbaryl resistance is monogenic, and genetic evidence suggests that it may be associated with a variant of the vesicular acetylcholine transporter protein. Kinetics of inhibition of acetylcholinesterase from sensitive and resistant strains by carbaryl do not differ. This distinction is important for practical reasons, since optimal strategies for controlling the spread of monogenic resistance are quite different than are those best suited for containment of polygenic ones.

COST OF CROSS-RESISTANCE

It is difficult to estimate the cost of cross-resistance in economic terms. However, its occurrence can limit options with respect to insect control programs. The tobacco budworm, *Heliothis virescens*, was controlled with DDT in the 1950s and early 1960s, and resistance became widespread. The primary mechanism involved is knockdown resistance (*kdr*), which is now known to result from particular mutations in the voltage-gated sodium channel gene that confer insensitivity to both DDT and pyrethroids. Thus, cross-resistance has the potential of reducing the value of pyrethroids, one of

the most widely used classes of pesticides, as a control agent. The same mechanism, indeed the same mutations, are responsible for pyrethroid and DDT resistance in house flies and German cockroaches; hence, the problems associated with cross-resistance are not restricted to one species.

Cross-resistance may in some cases limit the usefulness of abamectin and related compounds; in the LPR strain of houseflies that is resistant to pyrethroids, resistance to abamectin has also been reported. This resistance appears to be metabolic in nature, the result of elevated levels of expression of a particular cytochrome P450 isoform. Interestingly, although both cyclodienes and abamectin have the same target site (the GABA receptor), cross-resistance between these two compounds has not been reported.

FUTURE PROSPECTS

As is the case with pesticide resistance in general, a number of developments show promise with respect to the management of cross-resistance in agricultural pests. Integrated pest management strategies use multiple control agents with differing modes of action in combination with other non-chemical methods of pest control. The objectives in these programs include minimizing the probability of resistance developing to a particular agent, and avoiding use of agents with similar modes of action that would lead to the development of cross-resistance.

Resistance management is a strategy that includes among its goals reduction in pesticide usage and resistance and cross-resistance management. It consists of using carefully designed methods to control levels of pest insect populations. As a control program, it represents a paradigm shift from the traditional goal of pest eradication to one of population (and thus cost) management. An example of this approach is the program used in Australia for the control of *Helicoverpa armigera*, in which timed pyrethroid and endosulfan applications, use of transgenic crops expressing *Bacillus thuringiensis* toxin, and maintenance of untreated refugia for sensitive insects are implemented on a national level to manage resistance.

Perhaps the most promising area with respect to reducing problems associated with cross-resistance is in the development of new control agents with completely novel modes of action. The importance of this has been recognized for over a decade, but it has been only recently that our understanding of insect physiology has progressed to the extent that agents can be designed in such a way as to target specific unique pathways.

Insect growth regulators (IGRs) are one such class of compounds. Agents like juvenile hormone analogs

and chitin synthetase inhibitors act on target sites quite different from those of organophosphates, carbamates, and pyrethroids, and in some cases are more species-specific in their toxicity (e.g., tebufenozide). In the case of lufenuron, resistance has been observed in field populations of *Drosophila* and has been postulated to be the result of cross-resistance to propoxur. However, the lack of correlation between levels of propoxur and lufenuron resistance in different fly strains suggests that this is a case of multiple-resistance rather than cross-resistance. These same strains are not resistant to cyromazine, another IGR.

In summary, the problem of cross-resistance can be viewed as a subset of the larger problem of pesticide resistance management. It is, however, a problem that new advances in genetics and drug design may be able to address. It is difficult to predict how resistance to a given compound may evolve—will it involve target site modification, altered metabolism, or a combination of the two? Compounds with novel target sites and/or pathways for metabolic degradation, in addition to offering the promise of increased selectivity in toxicity, also should be less limited in their application by patterns of pesticide resistance and thus possible cross-resistance that evolved as a result of the prior use of chemical control agents. Recent advances in medical genetics and pharmacology provide a model for the development of agents that are carefully targeted toward specific physiological processes. These same approaches may lead to the development of suites of pesticides with specific but diverse modes of action, so that problems associated with cross-resistance can be minimized.

BIBLIOGRAPHY

- Bull, D.L.; Menn, J.J. Strategies for managing resistance to insecticides in *Heliothis* pests of cotton. In *Managing Resistance to Agrochemicals: From Fundamental Research to Practical Strategies*; Green, M.B., LeBaron, H.M., Moberg, W.K., Eds.; American Chemical Society: Washington, DC, 1990; 118–133.
- Casida, J.E.; Quistad, G.B. Golden age of insecticide research: past, present, or future. *Annu. Rev. Entomol.* **1998**, *43*, 1–16.
- Clark, J.M.; Scott, J.G.; Campos, F.; Bloomquist, J.R. Resistance to avermectins: extent, mechanisms and management implications. *Annu. Rev. Entomol.* **1994**, *40*, 1–30.
- Committee on Strategies for the Management of Pesticide Resistant Pest Populations. *Pesticide Resistance: Strategies and Tactics for Management*; National Academy of Sciences: Washington, DC, 1986.
- Dhadialla, T.S.; Carlson, G.R.; Le, D.P. New insecticides with ecdysteroidal and juvenile hormone activity. *Annu. Rev. Entomol.* **1998**, *43*, 545–569.

- Forrester, N.W.; Bird, L.J. The need for adaptation to change in insecticide resistance management: The Australian experience. In *Molecular Genetics and Evolution of Pesticide Resistance*; Brown, T.M., Ed.; American Chemical Society: Washington, DC, 1995; 160–168.
- Scott, J.G. Investigating mechanisms of insecticide resistance: methods, strategies and pitfalls. In *Pesticide Resistance to Arthropods*; Roush, R.T., Tabashnik, B.E., Eds.; Chapman & Hall: New York, 1990; 39–57.
- Scott, J.G. Cytochromes *P450* and insecticide resistance. *Insect Biochem. Mol. Biol.* **1999**, *29* (9), 757–777.
- Wilson, T.G. Cyromazine toxicity in *Drosophila melanogaster* (Diptera: Drosophilidae) and lack of cross-resistance in natural population strains. *J. Econ. Entomol.* **1997**, *90* (5), 1163–1169.
- Wilson, T.G.; Cain, J.W. Resistance to the insecticides lufenuron and propoxur in natural populations of *Drosophila melanogaster* (Diptera: Drosophilidae). *J. Econ. Entomol.* **1997**, *90* (5), 1131–1136.
- Zlotkin, E. The insect voltage-gated sodium channel as a target of insecticides. *Annu. Rev. Entomol.* **1999**, *44*, 429–456.

Cruciferous Root Crop Insects: Ecology and Control

Stan Finch

Rosemary H. Collier

Warwick HRI, The University of Warwick, Wellesbourne, Warwick, U.K.

INTRODUCTION

Members of the plant family Cruciferae are found chiefly in north temperate regions. The Cruciferae, named after their flowers which bear four petals in the shape of a cross, are characterized by chemicals known as glucosinolates, which give the plants their characteristic tastes and odors.^[1] The glucosinolates are toxic to most insects, but certain species have adapted to them and now feed exclusively on one or more of the 220 plant genera found within the Cruciferae.^[2,3]

The two genera of Cruciferae cultivated most extensively are *Brassica* L. and *Raphanus* L. The main cruciferous “root” (really swollen stem bases) crops are turnips (*Brassica campestris* L. var. *rapifera* Metz.), swedes or rutabagas [*B. napus* L. var. *napobrassica* DC. (L.) Reichenb], radish (*R. sativus* L. var. *radicula* Person), and Chinese/Japanese radish (*R. sativus* L. var. *longipinnatus* Bailey), often called “mooli.”

In temperate regions, cruciferous root crops can be attacked by 50–60 insect species. The relative importance of individual pest species varies from crop to crop and from country to country. Pest management involves ensuring that the crop plants 1) are not killed during establishment; 2) do not suffer too much foliar damage during growth; and 3) that their “roots” are kept relatively damage free.

PEST INSECT ECOLOGY

Pests Affecting Seedling Establishment

The seeds of cruciferous root crops are drilled directly into the soil. For crops of bunching radish, used mainly in salads, the seeds are sown at a high density. In most crops that produce large roots, however, the seed is often drilled to produce an extremely precise stand in which, depending on crop, the individual plants are spaced from 5 to 25 cm apart. Without adequate protection at this stage, even relatively few pest insects can soon create patchy crops.

Flea beetles are the most troublesome insects during seedling establishment. The adult beetles are 1.5–3.0 mm long and are characterized by their enlarged hind

femora (Graphic 1) with which they make long “flea-like” leaps. Most flea beetles that damage cruciferous root crops belong to the genus *Phyllotreta*. The species are described in detail in Ref.^[4] and can be separated by color into ones resembling *P. cruciferae*, in which the elytra are of one color and have a metallic luster, and ones resembling *P. nemorum*, in which the elytra are black with two longitudinal yellow bands (Fig. 1).

The adult beetles overwinter in plant debris alongside field boundaries. They become active in the spring and disperse to find new crops generally when midday temperatures rise above 20°C. Once a crop is located,^[5] the beetles settle and start to chew either on the plant stem below the soil surface or on the cotyledons. Such damage gives the seedlings a characteristic “shot-hole” appearance.

Foliar Pests

Plant foliage can be damaged by both aphids and caterpillars.

The cabbage aphid (*Brevicoryne brassicae*), which is the main pest aphid, remains on herbaceous Cruciferae throughout its life cycle.^[6] This aphid overwinters as the parthenogenetic adult or, in more northern areas, as the egg stage. In the spring, alatae disperse to find new host plants on which further batches of apterae and, later, alatae are produced parthenogenetically. *B. brassicae* colonizes swedes/rutabagas and radish but not turnips and causes problems mainly in warm dry years, when conditions are favorable for the aphid to produce large colonies.

A second pest aphid, the turnip aphid [*Lipaphis erysimi* (Kalt.)], occurs usually on weeds in Europe but is a serious pest of crops in Asia and parts of the New World. The biology of this aphid is similar to that of the cabbage aphid. It differs by producing considerably less wax and by causing the plant leaves to curl and form small pockets in which the aphids live.

Caterpillars of several species of Lepidoptera (Fig. 2) damage the foliage of cruciferous root crops.^[7] Caterpillars of the small white butterfly (*Pieris rapae*), the “imported cabbage worm” of the U.S.A. and Canada, are a major problem in cruciferous leafy crops (e.g., cabbage, cauliflower) in most countries.^[8] They are



Fig. 1 Adults of the flea beetles *P. cruciferae* and *P. nemorum* (striped elytra).

not as important in root crops, however, largely because this butterfly lays its eggs singly. Hence, if only small amounts of caterpillar damage occur on most plants, the damage can be tolerated. In contrast, the large white butterfly (*Pieris brassicae*) deposits all of its eggs on just one plant and so its infestations are localized. This species now rarely merits widespread control in Western Europe, but in Eastern Europe, where it still occurs in high numbers, it remains a major pest. This species has not yet reached the U.S.A. and Canada. Species such as the diamond-back moth (*Plutella xylostella*) and the cabbage moth (*Mamestra brassicae*) cause considerable damage in continental Europe, where the higher summer temperatures enable them to complete more generations than in countries further north. Damage by

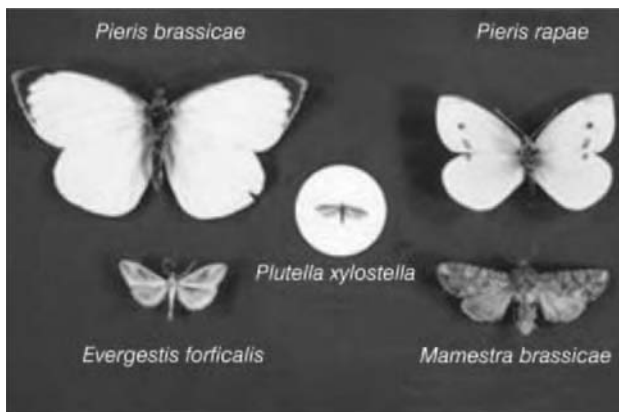


Fig. 2 Five species of Lepidoptera whose caterpillars damage the foliage of cruciferous root crops in the U.K.

the garden pebble moth (*Evergestis forficalis*) occurs mainly around the edges of crops.

Soil Pests

The major pest of the actual “roots” is the cabbage root fly (*Delia radicum* L.) (Fig. 3), or cabbage maggot (*Hylemya brassicae*), as the larvae of this fly damage the part of the plant used for human consumption. *D. radicum* occurs throughout the temperate zone of the holarctic region (35°–60°N) and can have up to five generations a year in southern Europe and the U.S.A. The females lay their eggs in the soil alongside the stems of cruciferous plants and, once the eggs hatch, the small larvae tunnel down through the soil to feed on the plant’s roots. Most larvae feed in the superficial layers of the “roots” (Fig. 4) but some tunnel deeper. Larvae of the turnip fly (*Delia floralis*) are often found feeding with those of *D. radicum* in northern parts of Canada and Europe and often displace *D. radicum* in the most northerly regions.

BIOLOGICAL CONTROL

Predators

Populations of most pest insects are regulated naturally by predators and parasitoids. The well-known predators of aphids are lady-beetles (Coccinellidae) and hover-flies (Syrphidae). Caterpillars of the butterflies and moths are eaten by birds and predatory ground beetles. Most predators are opportunistic feeders and so tend to eat those pest insects that are



Fig. 3 Female cabbage root fly feeding from the flowers of the umbel *Heracleum sphondylium*.



Fig. 4 Swede “root” damaged by larvae of the cabbage root fly.

present in highest numbers. However, as no predator feeds specifically on just one pest insect species, once the original prey become scarce the predators either leave the fields to find higher densities of a preferred prey or simply start to feed on alternative prey. As a result, predators rarely kill sufficient pest insects to prevent crop damage.

Parasitoids

The commonest parasitoids of the caterpillars of the two Pierid butterflies are the wasps *Apanteles glomeratus* (Hymenoptera: Braconidae), which kill the caterpillars as they are about to pupate, and *Pteromalus puparum* (Hymenoptera: Pteromalidae), which destroy the pupae. Similarly, the larvae of the cabbage root fly are parasitized by the wasp *Trybliographa rapae* (Hymenoptera: Eucoilidae) and the pupae of the fly by the beetle *Aleochara bilineata* (Coleoptera: Staphylinidae).^[9] As natural control by parasitoids usually acts late in the life cycle of the pest it rarely reduces pest damage on existing crops. However, high

parasitoid activity does ensure that fewer pest insects survive to damage subsequent crops.

CULTURAL CONTROL

Many of the pest insects of cruciferous plants are capable of dispersing thousands of meters in flight and so, on an individual-farm scale, the use of rotation to isolate crops is often impractical. It is widely believed also that wild host plants serve as bridges that prevent effective isolation. However, for *D. radicum* such arguments are not convincing, as although this fly can develop on a wide range of wild cruciferous plants, it is rarely found on such species in the field. The major sources of pest insects are the previous cruciferous crops. It has been suggested that crop rotation might be effective if it could be implemented on a regional basis. Even this now seems unlikely, as the introduction of oilseed rape (“Canola” in Canada), as a break crop in the production of cereals, means that most countries in the northern hemisphere now grow much larger hectares of cruciferous oilseed crops (mainly cultivars of *B. napus* L. var. *oleifera* (E. & G.) and *B. campestris* L. var. *rapifera* Metz. Ssp. *oleifera* Metz.) than cruciferous vegetable crops, of which cruciferous root crops form only a small part. Control problems in cruciferous root crops have been exacerbated in recent years, as the small amounts of insecticide applied to oilseed crops means that such crops have now become a major source of pest insects.

RESISTANT PLANTS

Attempts to select cultivars resistant to the cabbage root fly have not been too successful. Much of the “resistance” found was based on the females preferring not to lay eggs on certain cultivars (antixenosis) rather than on killing the feeding larvae (antibiosis). In addition, resistant cultivars on their own would need extremely high levels of “resistance” to keep the roots damage free. Nevertheless, the seed companies now avoid highly susceptible breeding lines and select those with some level of resistance to the major pest insects, as even partial resistance helps to improve the effectiveness of insecticidal control.

INSECTICIDAL CONTROL

At present many insecticidal products are available for controlling flea beetles, aphids, and caterpillars feeding on the aerial parts of plants.^[10] The 25 products approved for this purpose in the U.K. contain one of just four active ingredients, deltamethrin, pirimicarb,

nicotine, and rotenone. The two chemicals applied to the majority of crops are the general insecticide deltamethrin (Pyrethroid) and the aphicide, pirimicarb (Carbamate). Should any of the above chemicals be withdrawn, or any insect develop resistance to them, there are now new active ingredients that would probably be approved as replacements.

Flea beetle damage can occur before many seedlings emerge from the soil, and so the best strategy is to apply insecticide either to the seed or to the soil at drilling. Unfortunately, pyrethroid insecticides are not effective when applied to the soil. However, when beetle numbers are high, pyrethroid insecticides can be applied after the seedlings have emerged from the soil. The latter is often imperative in radish grown for bunching, as the plants are sold with the foliage left on.

Insecticides are applied against foliar pests only when aphid numbers are high or caterpillar damage becomes clearly evident. It is important to prevent too much leaf damage, as the weight of the plant foliage reflects the final weight (yield) of the harvested roots. In crops used for human consumption, the number of marketable units per hectare is more important than total weight (yield). High yield is important in crops grown for animal feed and is achieved largely by growing cultivars selected specifically for the purpose.

At present, the major concern is how to prevent damage to the subterranean parts of cruciferous root crops.^[5] Apart from the impact of flea beetles on seedling emergence, controlling the cabbage root fly is now the major problem in the U.K. This situation has arisen because the organophosphorus insecticide used for the past 40 years to control this pest, chlorfenvinphos, can no longer be used to kill the fly larvae in swede crops, and there is no effective alternative. Unfortunately, even when confined on newly sprayed foliage, the adult of this fly is not killed by deltamethrin or any of a wide range of other pyrethroid insecticides. Hence, to obtain the damage-free roots demanded by the supermarkets, producers now have to grow their crops under the light-weight mesh covers developed originally for extending the growing season in other vegetable crops.

CONCLUSIONS

There is an urgent need to find an insecticide to kill the cabbage root fly. At present, the grower's only options for roots needed for human consumption are to grow the crop in the open and select and trim the least damaged roots or to grow the crop under covers.

REFERENCES

1. Finch, S. Chemical attraction of plant-feeding insects to plants. In *Applied Biology 5*; Coaker, T.H., Ed.; Academic Press: London, 1980; 67–143.
2. Finch, S. Horticultural crops. In *Integrated Pest Management*; Burn, A.J., Coaker, T.H., Jepson, P.C., Eds.; Academic Press: London, 1987; 257–293.
3. Finch, S.; Thompson, A.R. Pests of cruciferous crops. In *Vegetable Crop Pests*; McKinlay, R.G., Ed.; Macmillan Press: London, 1992; 87–138.
4. Hill, D.S. *Agricultural Insect Pests of Temperate Regions and Their Control*; Cambridge University Press: Cambridge, 1987.
5. Finch, S.; Collier, R.H. Host-plant selection by insects—a theory based on ‘appropriate/inappropriate landings’ by pest insects of cruciferous plants. *Entomol. Exp. Appl.* **2000**, *96*, 91–102.
6. Blackman, R.L.; Eastop, V.F. *Aphids on the World Crops—An Identification and Information guide*; John Wiley & Sons Ltd.: Chichester, England, 2000.
7. Gratwick, M., Ed.; *Crop Pests in the UK—Collected Edition of MAFF Leaflets*; Chapman & Hall: London, 1992.
8. Jones, F.G.W.; Jones, M.G. *Pests of Field Crops*; Edward Arnold Ltd.: London, 1977.
9. Finch, S. A review of the progress made to control the cabbage root fly (*Delia radicum*) using parasitoids. In *Arthropod Natural Enemies in Arable Land, II, Survival, Reproduction and Enhancement*; Aarhus University Press: The Netherlands, 1996; 227–239.
10. Kennedy, R.; Collier, R. Pests and diseases of field vegetables. In *Pest and Disease Management Handbook*; Alford, D.V., Ed.; Blackwell Science: Oxford, 2000; 185–257.

Defoliants for Cotton

C. O. Gwathmey

C. C. Craig Jr.

Department of Plant Sciences, University of Tennessee, Jackson, Tennessee, U.S.A.

INTRODUCTION

Defoliants and desiccants are types of harvest-aid chemicals used in cotton (*Gossypium* spp.) production. The most commonly cultivated species is upland cotton (*Gossypium hirsutum* L.). Defoliants are applied to the crop to cause leaves to drop from plants in preparation for harvest. Desiccants are also used to prepare for cotton harvest, but their main purpose is to dry plant material rapidly.

Prior to widespread adoption of mechanical harvesting, there was little interest in defoliating cotton, because contamination from foliage was minimal during hand harvesting. For mechanical harvest, however, the remaining foliage can reduce picker efficiency, add trash, and cause discoloration of the lint.^[1] Chemical defoliation prior to harvest allows the crop to be harvested efficiently, while yield and fiber quality are at their peak. The cleanliness of cotton at the gin and textile mill and the value of the lint are improved by defoliation. Another benefit is reduced moisture content in harvested lint and seed—essential to storage of seedcotton in modules.^[2]

Defoliants are commonly used in cotton production areas where spindle pickers are used for harvesting the crop. Desiccants are more commonly used where stripper-type harvesters are used.^[3] This type of harvester requires dry plant material to operate efficiently and to optimize cotton quality.

CATEGORIES OF DEFOLIANTS BY MODE OF ACTION

Herbicidal defoliants injure the plant, causing it to produce ethylene in response. Ethylene promotes leaf abscission by increasing the activity of enzymes such as pectinase and cellulase, which degrade cell walls and middle lamellae in the abscission zone of the petiole.^[4] However, severe injury from herbicidal defoliants can cause leaves to die before they abscise. Dead leaves remaining on the plant contribute to trash in the harvested cotton.

Desiccants are relatively harsh types of herbicidal defoliants that disrupt membrane integrity, causing cells to lose water rapidly.^[4] At low use rates, however,

certain desiccants act as defoliants by the injury mechanism described above.

Hormonal defoliants enhance ethylene production and/or inhibit auxin transport in the plant. The balance of these hormones affects leaf abscission. Cells in the abscission layer in the petiole separate due to cell wall degrading enzymes that respond to decreasing auxin-to-ethylene ratio.^[4] Defoliation response of hormonal defoliants is generally more sensitive to temperature and crop conditions than that of herbicidal defoliants (Table 1).

Defoliants

Carfentrazone-ethyl^[5] is an herbicidal defoliant that inhibits an enzyme (protoporphyrinogen oxidase, or PPO) essential to chlorophyll biosynthesis. This inhibition results in accumulation of reactive oxygen species in leaf cells, causing peroxidation of membrane lipids and loss of membrane integrity and turgor.^[4] This defoliant may act as a desiccant at high use rates.

Dimethipin was commercially introduced in the 1980s.^[1] This defoliant causes a loss of stomatal control of transpiration, leading to gradual loss of leaf turgor.^[6] This response induces the release of ethylene in the plant. Dimethipin may be considered either a herbicidal or hormonal defoliant.

Pyraflufen-ethyl is a herbicidal defoliant similar to carfentrazone-ethyl. This PPO inhibitor may act as a desiccant at high use rates.

Thidiazuron was commercially introduced in the 1980s.^[1] Thidiazuron inhibits the polar transport of auxins in the plant, decreasing the auxin-to-ethylene ratio and inhibiting regrowth of foliage.^[7] This hormonal type of defoliant is relatively effective in removing immature leaves. Its defoliation effectiveness is diminished under cooler conditions. Therefore, it is frequently mixed with other harvest aids^[8] or adjuvants.^[9]

Thidiazuron and diuron are available commercially as a prepackaged mixture. The herbicidal action of diuron is intended to increase defoliation activity under cooler conditions, relative to thidiazuron alone, but it can cause desiccation under warm conditions at high use rates.^[8]

Tribufos was introduced in the 1960s.^[1] Tribufos is a herbicidal defoliant that injures the palisade cells of

Table 1 Defoliant and other harvest aids commonly used in commercial cotton production

| Type | Common name | Chemical name |
|-----------------------|------------------------|--|
| Defoliant | Carfentrazone-ethyl | Ethyl 2-dichloro-5-[4-(difluoromethyl)-4,5-dihydro-3-methyl-5-oxo-1H-1,2,4-triazol-1-yl]-4-fluorobenzenepropanoate |
| Defoliant | Dimethipin | 2,3-Dihydro-5,6-dimethyl-1,4-dithiin-1,1,4,4-tetraoxide |
| Defoliant | Pyraflufen-ethyl | Ethyl 2-chloro-5-(4-chloro-5-difluoromethoxy-1-methyl-1H-pyrazol-3-yl)-4-fluorophenoxyacetate |
| Defoliant | Thidiazuron | <i>N</i> -Phenyl- <i>N'</i> -1,2,3-thidiazol-5-ylurea |
| Defoliant | Thidiazuron + diuron | <i>N</i> -Phenyl- <i>N'</i> -1,2,3-thidiazol-5-ylurea + 3-(3,4-dichlorophenyl)-1,1-dimethylurea |
| Defoliant | Tribufos ^a | <i>S,S,S</i> -Tributyl phosphorotrithioate |
| Boll opener | Ethephon | (2-Chloroethyl)phosphonic acid |
| Boll opener/defoliant | Ethephon + AMADS | (2-Chloroethyl)phosphonic acid + 1-aminomethanamide dihydrogen tetraoxosulfate |
| Boll opener/defoliant | Ethephon + cyclanilide | (2-Chloroethyl)phosphonic acid + 1-(2,4-dichlorophenylamino carbonyl)-cyclopropane carboxylic acid |
| Desiccant | Paraquat | 1,1'-Dimethyl-4,4'-bipyridinium dichloride |
| Desiccant | Sodium chlorate | Sodium chlorate |

^aOther common names are butifos, merphos, and tribufate. (From Ref.^[4].)

leaves, causing the plant to generate ethylene as a stress response.^[4] High use rates can cause leaf desiccation.

Boll Opener/Defoliants

Ethephon is a precursor to ethylene, which serves to generate ethylene in the plant. It is mainly used to promote boll opening in cotton production, but it also enhances defoliation. Because its hormonal effects diminish under cool temperatures, ethephon is commonly mixed with other harvest aids to improve its defoliation effects.^[8] Low rates of ethephon may be used to “condition” the crop by increasing plant ethylene concentration prior to defoliant application.^[1]

Ethephon and 1-aminomethanamide dihydrogen tetraoxosulfate (AMADS) are commercially available as a prepackaged mixture. AMADS is an ethylene synergist intended to improve defoliation response.^[4]

Ethephon and cyclanilide are commercially available as a prepackaged mixture. Together, they have hormonal defoliation and boll opening effects.^[10] Cyclanilide is an auxin transport inhibitor. The combination of ethephon and cyclanilide decreases auxin relative to ethylene concentration in the plant, thus enhancing cellulase activity in the leaf abscission layer. Cyclanilide also inhibits terminal regrowth.^[10]

Desiccants

Paraquat is used mainly as a desiccant. It causes the plant to generate free radicals that disrupt cell membranes, leading to a rapid loss of moisture.^[4]

Sodium chlorate is a strong oxidizing agent. It acts as a defoliant at relatively low application rates and as a desiccant at higher rates. Sodium chlorate remains popular in areas where low yields do not justify use of costlier harvest aids, or where restricted-use materials cannot be applied.^[1] In some areas, it is applied as a defoliant under cool weather conditions prevalent in late season.

APPLICATION METHODS AND PRECAUTIONS

Defoliants are typically applied to the crop in aqueous solution through a ground-based or an aerial spray system.^[11] Application efficiency is influenced by spray droplet size and placement. Droplet size is largely a function of spray pressure, nozzle type, and use of adjuvants. Larger droplets are less prone to aerial drift, but smaller droplets may be distributed more uniformly in the leaf canopy. Thorough spray coverage is essential for satisfactory defoliation, because most harvest aids are not translocated within the plant. More than one application may be necessary where the crop canopy is so dense that a single application is inadequate.

Minimizing nontarget drift is an important objective of spray technology development. New technologies that have improved defoliation efficiency include air-induction nozzles that produce larger droplets that are less susceptible to drift.^[11] Placement of droplets is largely determined by the type and operation of the spray equipment. Ground-based, high-clearance

sprayers are typically used to defoliate relatively small or irregular fields that may be difficult to treat by aircraft, given the need to prevent non-target application. Aerial application is typically used in large fields that would be difficult to cover using ground-based sprayers.

Crop response to defoliant varies in response to unpredictable changes in weather conditions during or after application.^[5] To improve the likelihood of a favorable response, producers often apply a mixture of harvest aids with different properties.^[12] For instance, a herbicidal defoliant such as tribufos may be added to hormonal materials such as ethephon and cyclanilide, to improve defoliation response under cool conditions. Producers may also add an adjuvant to the spray solution in an effort to improve droplet performance and plant uptake of active ingredients.^[9]

CONCLUSIONS

Efficient application of appropriate defoliant improves return on harvest aid investment, economic value of the cotton crop, and protection of non-target vegetation. Like all agricultural chemicals, however, defoliant must be used in accordance with product label guidelines and effective environmental stewardship practices.

REFERENCES

1. Crawford, S.H.; Cothren, J.T.; Sohan, D.E.; Supak, J.R. A history of cotton harvest aids. In *Cotton Harvest Management: Use and Influence of Harvest Aids*; Supak, J.R., Snipes, C.E., Eds.; The Cotton Foundation: Memphis, TN, 2001; 1–20.
2. Brashears, A.D.; Keeling, J.W.; Valco, T.D. Storage of seed cotton after four harvest aid treatments. Proceedings Beltwide Cotton Conferences, New Orleans, LA, Jan 6–10, 1997; National Cotton Council: Memphis, TN, 1997; 1609–1612.
3. Supak, J.R. Harvest aids for picker and stripper cotton. Proceedings Beltwide Cotton Conferences, San Antonio, TX, Jan 4–7, 1995; National Cotton Council: Memphis, TN, 1995; 5–8.
4. Cothren, J.T.; Gwathmey, C.O.; Ames, R.B. Physiology of cotton defoliation and desiccation. In *Cotton Harvest Management: Use and Influence of Harvest Aids*; Supak, J.R., Snipes, C.E., Eds.; The Cotton Foundation: Memphis, TN, 2001; 21–50.
5. Gwathmey, C.O.; Cothren, J.T.; Legé, K.E.; Logan, J.; Roberts, B.A.; Supak, J.R. Influence of environment on cotton defoliation and boll opening. In *Cotton Harvest Management: Use and Influence of Harvest Aids*; Supak, J.R., Snipes, C.E., Eds.; The Cotton Foundation: Memphis, TN, 2001; 52–72.
6. Metzger, J.D.; Keng, J. Effects of dimethipin, a defoliant and desiccant, on stomatal behavior and protein synthesis. *J. Plant Growth Regul.* **1984**, *3*, 141–156.
7. Suttle, J.C. Disruption of the polar auxin transport system in cotton seedlings following treatment with the defoliant thidiazuron. *Plant Physiol.* **1988**, *86*, 241–245.
8. Brecke, B.J.; Banks, J.C.; Cothren, J.T. Harvest-aid treatments: products and application timing. In *Cotton Harvest Management: Use and Influence of Harvest Aids*; Supak, J.R., Snipes, C.E., Eds.; The Cotton Foundation: Memphis, TN, 2001; 119–142.
9. Snipes, C.E.; Wills, G.D. Influence of temperature and adjuvants on thidiazuron activity in cotton leaves. *Weed Sci.* **1994**, *42*, 13–17.
10. Pedersen, M.K.; Burton, J.D.; Coble, H.D.; Collins, J.R.; Fritz, D. Efficacy of finish and its mechanism of action. Proceedings Beltwide Cotton Conferences, New Orleans, LA, Jan 6–10, 1997; National Cotton Council: Memphis, TN, 1997; 1363–1365.
11. Bader, M.J.; Sumner, P.E.; Culpepper, S. Harvest-aid application technology. In *Cotton Harvest Management: Use and Influence of Harvest Aids*; Supak, J.R., Snipes, C.E., Eds.; The Cotton Foundation: Memphis, TN, 2001; 143–166.
12. Snipes, C.E.; Cathey, G.W. Evaluation of defoliant mixtures in cotton. *Field Crops Res.* **1992**, *28*, 327–334.

Delusory Parasitosis

Nancy Hinkle

Department of Entomology, University of Georgia, Athens, Georgia, U.S.A.

INTRODUCTION

Delusory parasitosis (DP, also called “delusions of parasitosis,” “psychogenic parasitosis,” and “Ekbom’s syndrome”) is the conviction that one’s body is infested with microscopic or invisible creatures (insects, mites, worms, etc.).^[1,2] The individual may perceive these parasites biting, stinging, burrowing, or crawling into, over, or out of the skin. Frequently the sufferer’s home, automobile, and other inanimate surroundings are considered infested as well. According to the psychiatric community, this delusion of being parasitized is the most common of delusional beliefs^[3] but, because most cases are directed to entomologists or pest control services, and not perceived as a medical condition, this syndrome is much more common than is documented in the medical literature.^[4]

WHAT ARE THE SYMPTOMS OF DELUSORY PARASITOSIS?

DP sufferers’ accounts share many commonalities. These “bugs” are said to change color, appear and disappear before one’s eyes, invade and exit from body orifices, enter the skin and reappear, and persist despite repeated treatments of the body and environment.^[2] Apparently these creatures are facultative parasites, because they are said to be able to survive on inanimate (and inorganic) material, such as furniture and automobiles, as well as to infest and colonize human bodies.^[1,2]

While sufferers admit that no one else can see the pest, they display scarified lesions on their bodies as evidence of infestation, with self-mutilation ranging from scratches to deep ulceration.^[5] They also provide gobbets of tissue dug out of their skin and other debris for microscopic examination. Samples typically include lint, dandruff and scurf, scabs and dried blood, bits of skin (or oral mucosa, if infestation is perceived in the mouth), dirt, and miscellaneous debris.^[6] In addition to submitting material extracted from their skin, sufferers frequently supply debris dusted from windowsills and vacuum cleaner bags full of sweepings. They provide a detailed history of their condition, with elaborate descriptions of the pest and its life cycle. Typically, they have conducted extensive research and

are convinced they know what the causative agent is, requiring only confirmation from the entomologist.^[7] Despite never having traveled outside the United States, they are convinced they are infested by human bots or guinea worms, for instance. Frequently, family members or coworkers suffer from similar symptoms, which they consider evidence of a valid infestation.

Although they typically call the pests “invisible,” sufferers provide detailed descriptions, with recurrent themes.^[2] Generally the insects are black and white, but change colors, and are clear. They lay eggs that hatch into larvae and burrow into the skin. They are most active at night and can crawl, fly, and hop, often producing sticky fibers.^[8]

By the time the DP sufferer finds an entomologist, the “infestation” often has persisted for months or years and they have visited numerous physicians, including specialists such as dermatologists.^[5] They have been prescribed scabicides, louse shampoos, and antibiotics, some of which have provided temporary relief via the placebo effect, but none of which has solved the problem. It is not uncommon to find that they have self-medicated with herbal remedies and veterinary parasiticides.^[9] They have had their home treated repeatedly by pest control companies and frequently have applied over-the-counter pesticides to the home and vehicles.^[2] While they state emphatically that they are not crazy, their first words are often that they are “desperate,” and “you are my last hope.”

The typical sufferer is an older female, but individuals of all ages who recently have experienced a traumatic life event (job loss, bereavement, etc.) and are socially isolated are disproportionately likely to experience DP symptoms. The classic DP case is cited as Traver,^[8] illustrating that even highly educated scientists can suffer under the misapprehension that their body is infested with unknown organisms. This University of Massachusetts zoologist spent years examining samples from her scalp, ultimately describing a “new” mite, later determined to be the common house dust mite—a normal contaminant of the swabs she was using to procure samples.^[10]

Responses to DP symptoms can be quite extreme. Sufferers commonly report treating their bodies with household cleansers and other harsh chemicals; bathing in gasoline,^[1] ammonia, alcohol, kerosene, and pesticides;^[2,8,11] shaving the hair from their scalp or entire

body;^[11] and self-medicating with herbal medications, home remedies, and veterinary drugs.^[9] They spend inordinate amounts of time in house-cleaning, boiling their clothing and bed linen,^[1] and discarding or destroying “infested” furnishings.^[7] Not uncommonly, they have abandoned or euthanized their pets, assuming that their “infestation” originated from the animals.^[4,8,12] Many have moved out of their homes and are living in motels or their automobiles.^[4,11,13]

Although often these individuals are above average in intelligence and some are highly educated,^[1] no evidence to the contrary can convince them that they are not infested. A delusion is “a false belief that is not consistent with the patient’s intelligence, educational level, or cultural background, and that cannot be corrected by reasoning,”^[3] so the doctor’s and entomologist’s inability to find the creature does not dissuade DP sufferers from their conviction.

WHAT CAUSES DELUSORY PARASITOSIS SYMPTOMS?

The underlying cause of DP is unknown, but potential sources include physical, psychological, and physiological agents.^[13] Once arthropods have been excluded as the cause, these other possibilities should be explored.

Physical elements include exposure to irritants such as rock wool (found in acoustical ceiling tiles and insulation), fiberglass (found in insulation and some industrial fabrics), formaldehyde (from construction materials), etc.^[1] Exceptional investigative skills are needed to track down causative agents in situations such as would result from mixing fiberglass curtains with bed linen or clothing in the washing machine, dislodging fiberglass threads to contaminate the clothing, causing itching and irritation.

Psychological causes include depression, stress, anxiety, and a range of other mental or emotional conditions.^[3,6,11] According to psychological literature, DP may be initiated by primary tactile stimulus, real or imagined, producing pruritus, urticaria, paresthesia, or other sensations. In attempting to identify the causative agent, the sufferer visualizes common objects (threads, scabs with entrapped hairs, other effluvia) as creatures and attributes the sensation to them.^[2,10,13]

Physiological factors that can precipitate DP symptoms include not only a range of diseases with dermatological manifestations, but many medications as well. Over a hundred medical conditions manifesting as urticaria, paresthesia, pruritus, erythema, and other DP symptoms have been chronicled,^[2,4,14] including diseases such as diabetes, hypothyroidism, hepatic disease, heavy metal poisoning, neoplasms, etc. Virtually all medications, over-the-counter and prescription, list

at least one likely DP symptom as a potential side effect,^[2] not to mention possible drug interactions.

Recreational drug use may account for many DP cases, because formication, a sensation of insects crawling on the skin, is a well-known effect of various hallucinogens.^[6] “Cocaine bugs” is a term used to describe the tactile and visual hallucinations that accompany illicit drug use, especially with drugs such as cocaine and methamphetamines.^[2]

WHAT IS THE ROLE OF ENTOMOLOGISTS IN DEALING WITH DELUSORY PARASITOSIS?

Entomologists are limited in what they can offer these people, but they can examine the proffered materials and determine whether there is evidence of arthropod involvement. In particular, they should exclude potential etiologic agents such as fleas, lice, bed bugs, rodent mites, bird mites, straw itch mites, thrips, etc.^[1,2] by surveying the home (using glueboards, vacuum, light traps, etc.) and examining specimens. Adhesive tape applied to the site as the sensation is experienced provides the most meaningful sampling technique because it represents the area of concern—the individual’s body.

Symptoms should not be brushed off, as they can be indicative of life-threatening medical conditions. In the same way that individuals experiencing heart attacks attribute their symptoms to heartburn, thereby forgoing timely medical interventions that could prevent death, the smokescreen of “invisible bugs” may delay diagnosis and treatment of underlying conditions. For instance, paresthesia is a common manifestation of transient ischemic attacks (“mini-strokes”) and various neuropathies.^[14] While assuring the sufferer that the symptoms are real and deserve further study, entomologists can encourage individuals to accept physician-prescribed medication to alleviate discomfort while pursuing investigation of possible underlying causes. It is important that DP sufferers realize that there are several valid possibilities for what is producing their symptoms, and that spraying insecticides will not solve the problem.

CONCLUSIONS

Delusory parasitosis is not an entomological problem, but it is Entomology’s problem. Entomologists spend thousands of hours annually listening to DP sufferers, examining samples, and attempting to help them find solutions. While professionally they are limited to determining whether an arthropod is involved, they may provide referrals to specialists such as environmental hygienists or physicians. DP sufferers typically are disinclined to consider psychological causations, responding

to such suggestions with hostility.^[5] Nevertheless, DP, whether physiological or psychological, is a medical situation, and entomologists should encourage sufferers to persist in their search for a physician willing to thoroughly investigate their condition.

REFERENCES

1. Ebeling, W. Delusory parasitosis and "cable mite" dermatitis. In *Urban Entomology*; Ebeling, W., Ed.; University of California Regents: Berkeley, CA, 1975.
2. Hinkle, N.C. Delusory parasitosis. *Am. Entomol.* **2000**, *46* (1), 17–25.
3. Koblenzer, C.S. The clinical presentation, diagnosis and treatment of delusions of parasitosis—a dermatologic perspective. *Bull. Soc. Vector Ecol.* **1993**, *18*, 6–10.
4. Sherman, M.D.; Holland, G.N.; Holsclaw, D.S.; Weisz, J.M.; Omar, O.H.M.; Sherman, R.A. Delusions of ocular parasitosis. *Am. J. Ophthalmol.* **1998**, *125*, 852–856.
5. Koo, J.; Lee, C.S. Delusions of parasitosis: a dermatologist's guide to diagnosis and treatment. *Am. J. Clin. Dermatol.* **2001**, *2* (5), 285–290.
6. Zanol, K.; Slaughter, J.; Hall, R. An approach to the treatment of psychogenic parasitosis. *Int. J. Dermatol.* **1998**, *37*, 56–63.
7. Goddard, J. Imaginary insect or mite infestations. *Infect. Med.* **1998**, *15* (3), 168–170.
8. Traver, J.R. Unusual scalp dermatitis in humans caused by the mite, *Dermatophagoides* (Acarina, Epidermoptidae). *Proc. Entomol. Soc. Wash.* **1951**, *53* (1), 1–25.
9. Freudenmann, R.W. Der "dermatozoenwahn." *Fortschr. Neurol.-Psychiatrie* **2002**, *70* (10), 531–541.
10. Poorbaugh, J.E. Cryptic arthropod infestations: separating fact from fiction. *Bull. Soc. Vector Ecol.* **1993**, *18* (1), 3–5.
11. Kim, C.; Kim, J.; Lee, M.; Kang, M. Delusional parasitosis as "folie a deux". *J. Kor. Med. Sci.* **2003**, *18*, 462–465.
12. Nel, M.; Schoeman, J.P.; Lobetti, R.G. Delusions of parasitosis in clients presenting pets for veterinary care. *J. S. Afr. Vet. Assoc.* **2001**, *72* (3), 167–169.
13. Le, L.; Gonski, P.N. Delusional parasitosis mimicking cutaneous infestation in elderly patients. *Med. J. Aust.* **2003**, *179* (4), 209–210.
14. Berrios, G.E. Delusional parasitosis and physical disease. *Comprehensive Psychiatry* **1985**, *26*, 395–403.

Dengue

Sushil Kumar Kabra
Tanu Singhal
Rakesh Lodha

Department of Pediatrics, All India Institute of Medical Sciences,
New Delhi, India

INTRODUCTION

Dengue, currently the most important arboviral disease of humans, is caused by any of the four serotypes of dengue virus; i.e., Den 1, 2, 3, and 4. The most important vectors are *Aedes aegypti* and *Aedes albopictus*.^[1]

The earliest scientific descriptions of dengue fever (DF) include epidemics of “knee fever in Cairo” and Batavia (Djakarta) in 1779 and Philadelphia in 1780.^[1] Since then, nearly all tropical and subtropical countries have reported outbreaks, and dengue is now endemic in all continents except Europe.^[2] Apart from geographical spread and rising disease incidence, the clinical picture since the 1950s has also changed from a benign disorder (classic dengue fever) to a serious disease with bleeding and shock [dengue hemorrhagic fever (DHF)]. Epidemic dengue hemorrhagic fever (DHF) occurs primarily in South Asia, the Americas, and some Pacific islands.^[1,2] Per current estimates more than 2.5 billion people live in dengue endemic areas of the world and approximately 100 million cases of dengue fever, 500,000 cases of dengue hemorrhagic fever, and 25,000 deaths occur annually.^[2]

PATHOGENESIS

The pathogenesis of DHF is not clearly established. It has been observed that DHF/dengue shock syndrome (DSS) usually occurs after sequential infection with any two of the four serotypes of dengue virus. It is postulated that primary infection with a particular serotype provides immunity only against the same serotype; a second infection with another serotype results in severe disease due to certain enhancing antibodies.^[3] However, as not all secondary infections lead to DHF, it has been proposed that variations in virulence and other biologic attributes of the virus also play a role in disease pathogenesis.^[4]

The major hemostatic abnormalities in DHF that differentiate it from DF are vasculopathy and coagulopathy, leading to plasma leakage, hemoconcentration, hypovolemia, and bleeding manifestations.^[5]

CLINICAL MANIFESTATIONS

Most dengue infections in young children are mild and indistinguishable from other common childhood febrile illnesses. Fever, headache, severe myalgia, arthralgia, skin rash, and malaise characterize classical dengue fever.^[6] Some patients with dengue fever have varying degrees of mucosal and cutaneous bleeding with mild thrombocytopenia without hemoconcentration or objective evidence of fluid leak in the form of ascites/pleural effusion. This phenomenon of dengue fever with unusual hemorrhage is seen during epidemics of DF and needs to be differentiated from DHF.^[6,7]

DHF can occur in all age groups but is most common in children less than 15 years old.^[1] Following an incubation period of 4–6 days, the illness usually begins abruptly with high fever accompanied by facial flushing and headache. Anorexia, vomiting, abdominal pain, and tender hepatomegaly are common, while splenomegaly is less frequent. All patients have some hemorrhagic phenomena in the form of a positive tourniquet test, petechiae, bruising at venepuncture sites, gum bleeds, epistaxis, hematemesis, or melena. The critical stage is reached when fever subsides after 2–7 days. The patient may then develop varying degrees of peripheral circulatory failure characterized by excessive sweating, restlessness, cool extremities, skin mottling, narrowing pulse pressure, hypotension, and eventually, irreversible shock. The unique feature of this disease progression is that circulatory failure is preceded by thrombocytopenia and a rise in hematocrit that can be detected by suitable laboratory tests. Unusual manifestations of DHF include hepatitis, encephalitis, and renal failure.^[6]

DIAGNOSIS

DF may mimic a wide variety of viral, bacterial, and rickettsial infections and is difficult to diagnose clinically. Differential diagnosis of DHF/DSS includes other viral hemorrhagic fevers, leptospirosis, Gram-negative sepsis, meningococemia, and typhoid. Falciparum

malaria can present with fever and bleeding and may be distinguished by the presence of splenomegaly and pallor.

The following criteria have been selected for clinical diagnosis of DHF:^[6]

- Clinical: Acute onset high-grade fever, hemorrhagic manifestations (at least a positive tourniquet test), shock.
- Laboratory: Thrombocytopenia (<100,000 per cubic mm), hemoconcentration (hematocrit elevated at least 20% above the standard for age, sex, and population or the baseline hematocrit), or other objective evidence of vascular leakage such as pleural effusion or ascites.

Two clinical criteria and one of the laboratory findings (or at least a rising hematocrit) are sufficient for making a provisional diagnosis of DHF. Presence of hypotension in a patient with a provisional diagnosis of DHF grades the disease as DSS.

LABORATORY FINDINGS

Laboratory findings in DHF include rising hematocrit, thrombocytopenia, and transformed lymphocytes on peripheral smear.^[5] While monitoring hematocrit, the possible effects of preexisting anemia, severe hemorrhage, or early volume replacement therapy should be kept in mind.^[6] There may be associated increased transaminases, hypoalbuminemia, hyponatremia, acidosis, and elevations in blood urea nitrogen and creatinine. In severe disease, there may be laboratory evidence of disseminated intravascular coagulation.^[5,6] X-ray film of the chest may reveal pleural-effusion commonly on the right side, occasionally bilateral. Abdominal ultrasound may detect thickened gall bladder wall with hepatomegaly and ascites. There may be electrocardiographic and echocardiographic abnormalities in some patients.^[8]

For confirmation of dengue virus infection, the virus may be isolated from blood during the early phase of illness. In the latter part (beyond 5 days), antibodies against the virus can be demonstrated by various methods such as hemagglutination-inhibition test (HI test) which detects both IgG and IgM antibodies, and IgM-capture enzyme linked immunosorbent assay (MAC-ELISA test), which measures dengue specific IgM.

TREATMENT

The treatment of DF is symptomatic. Fever is treated with paracetamol. Salicylates and other non-steroidal

anti-inflammatory drugs should be avoided as these may predispose a child to mucosal bleeds. In an epidemic setting, all patients with DF need regular monitoring by a primary care physician for early detection of DHF.

Patients with cold extremities, restlessness, acute abdominal pain, decreased urine output, or bleeding need urgent admission to a hospital, as do asymptomatic children with rising PCV and thrombocytopenia. Aggressive fluid replacement, supportive care, and intensive monitoring are the mainstay of treatment as no specific therapy is available. Intravenous fluids in the form of crystalloids (5% dextrose in Ringer's lactate/normal saline/half strength saline) or colloids (dextran 40) are administered initially in a rate and amount depending on the severity of the disease. Frequent monitoring of pulse rate, blood pressure, respiratory rate, central venous pressure, and packed cell volume is very crucial particularly in the early stages of the illness and guides further fluid therapy.^[9]

The role of plasma or platelet infusion in bleeding patients remains unclear. In a small study in which children with severe thrombocytopenia were included, platelet infusion did not alter the outcome of patients.^[10] In the presence of DIC, infusion of fresh frozen plasma and platelet concentrates may be beneficial. Fresh whole blood should be given to children with hypotension and low hematocrit. Steroids are of no benefit.^[11]

PROGNOSIS

The mortality in untreated DHF/DSS may be as high as 40–50%. Early recognition of illness, careful monitoring, and appropriate fluid therapy result in reduction in mortality to 1–5%.^[1] Recovery is fast (24–48 hr) and without sequelae.^[6] Presence of prolonged shock prior to intervention is associated with a very poor outcome and therefore emphasizes the need for early detection.

PREVENTION

In the absence of a safe and effective vaccine against dengue, vector control is at present the only way to prevent disease spread. Control of the adult mosquitoes by ultra-low volume (ULV) application of insecticides using aerial, ground, vehicle-mounted, and hand-carried equipment has been recommended particularly during epidemics.^[12] However, certain recent studies have demonstrated the transient and limited benefits of this approach.^[13,14] The relatively slower but more effective and sustainable methods are larval control measures.^[2] Elimination or cleaning of water-holding

containers that serve as the larval habitats of *A. aegypti* is important. Adding chemical larvicidal agents such as 1% temephos sand granules to stored water is very effective and has no ill effects. *Bacillus thuringiensis* H14 and larvivorous fish may be potentially useful biological larvicidal agents in the future.^[12]

Dengue has resurged as a major global public health problem and the current emphasis should be on vector control and appropriate case management strategies. Biological and social researches are essential to develop effective mosquito control measures, medications to reduce capillary leakage, and a safe tetravalent vaccine.

REFERENCES

1. Thongchareon, P.; Jatanesan, S. Epidemiology of dengue and dengue haemorrhagic fever. In *Monograph on Dengue/Dengue Haemorrhagic Fever*; World Health Organization: SEARO No. 22, New Delhi, 1993; 1–8.
2. Gubler, D.J. Dengue and dengue haemorrhagic fever. *Clin. Microbiol. Rev.* **1998**, *11* (3), 480–496.
3. Halstead, S.B. Dengue haemorrhagic fever. In *Hand book of Viral and Rickettsial Haemorrhagic Fevers*; Gear, J.H.S., Ed.; Boca Raton, FL, 1988; 85–94.
4. Rosen, L. The emperor's new clothes revisited, or reflections on the pathogenesis of dengue hemorrhagic fever. *Am. J. Trop. Med. Hyg.* **1977**, *26* (3), 337–343.
5. Srichaikul, T.; Nimmannitya, S. Haematology in dengue and dengue haemorrhagic fever. *Baillieres Best Pract. Res. Clin. Haematol.* **2000**, *13* (2), 261–276.
6. Nimmannitya, S. Clinical manifestations of dengue/dengue haemorrhagic fever. In *Monograph on Dengue/Dengue Haemorrhagic Fever*; World Health Organization: SEARO No. 22, New Delhi, 1993; 48–54.
7. Kabra, S.K.; Jain, Y.; Pandey, R.M.; Madhulika; Singhal, T.; Tripathi, P.; Broor, S.; Seth, P.; Seth, V. Dengue haemorrhagic fever in children in the 1996 Delhi epidemic. *Trans. R. Soc. Trop. Med. Hyg.* **1999**, *93* (3), 294–298.
8. Kabra, S.K.; Juneja, R.; Madhulika; Jain, Y.; Singhal, T.; Dar, L.; Kothari, S.S.; Broor, S. Myocardial dysfunction in children with dengue haemorrhagic fever. *Natl. Med. J. India* **1998**, *11* (2), 59–61.
9. Nimmannitya, S. Management of dengue and dengue haemorrhagic fever. In *Monograph on Dengue/Dengue Haemorrhagic Fever*; World Health Organization: SEARO No. 22, New Delhi, 1993; 55–61.
10. Kabra, S.K.; Jain, Y.; Madhulika; Tripathi, P.; Singhal, T.; Broor, S.; Dar, L.; Seth, V. Role of platelet transfusion in dengue haemorrhagic fever. *Indian Pediatr.* **1998**, *35* (5), 452–454.
11. Tassniyom, S.; Vaanawathan, S.; Chirawatkal, V.; Rojanasuphot, S. Failure of high dose methyl prednisolone in established dengue shock syndrome: a placebo controlled double blind study. *Pediatrics* **1993**, *92*, 111–115.
12. Bang, Y.H.; Tonn, R.J. Vector control and intervention. In *Monograph on Dengue/Dengue Haemorrhagic Fever*; World Health Organization: SEARO No. 22, New Delhi, 1993; 139–160.
13. Castle, T.; Amador, M.; Rawlins, S.; Figueroa, J.P.; Reiter, P. Absence of impact of aerial malathion treatment on *Aedes aegypti* during a dengue outbreak in Kingston, Jamaica. *Rev. Panam Salud Publica* **1999**, *5* (2), 100–105.
14. Perich, M.J.; Davila, G.; Turner, A.; Garcia, A.; Nelson, M. Behaviour of resting *Aedes aegypti* and its relation to ultra-low volume adulticide efficacy in Panama City, Panama. *J. Med. Entomol.* **2000**, *37* (4), 541–546.

Developing Cooperations: Pest and Pesticide Management

George Ekström

Swedish National Chemicals Inspectorate (KEMI), Solna, Sweden

Barbara Dinham

Eurolink Centre, Pesticide Action Network UK, London, U.K.

Henrik Kylin

Swedish University of Agricultural Sciences, Uppsala, Sweden

INTRODUCTION

The international community provides a number of instruments for mitigation of pesticide related problems in developing countries; e.g., the International Code of Conduct on Distribution and Use of Pesticides (the FAO Code), and the Basel, Rotterdam and Stockholm Conventions. Several United Nations' agencies and programs (individually or jointly as the IOMC) provide technical assistance. Most OECD member countries have programs to support sound management of pests and pesticides in the South. This article describes some of the problems (Figs. 1–4), selected international and bilateral activities, alternative approaches (Figs. 4–5, and gives recommendations to donors.

CONSTRAINTS TO PESTICIDE REDUCTION POLICIES IN DEVELOPING COUNTRIES

“Developing countries are the fastest growing pesticide markets, where health and environmental regulations are extremely limited, and a great deal of poisonings take place. Pesticide use is concentrated on export crops, such as cotton, fruit and vegetables.”^[1]

During a recent conference on safer pest management held in London, participants from developing countries identified the following constraints to pesticide reduction.^[2,3]

- Lack of impact assessments, effective pesticide policies, legislation, infrastructure (e.g., poisoning centers), registration and enforcement schemes, and lack of implementation of internationally agreed upon instruments.
- Lack of trained manpower, awareness campaigns, and partnership between public and private sectors.

- Lack of research and practical studies on alternatives, and reluctance of farmers to adapt alternatives.
- Inappropriate size of containers.
- Pesticide donations, and pressure from pesticide companies to keep products on the market.

INTERNATIONAL (OECD AND IOMC) COOPERATION FOR CAPACITY BUILDING

OECD Member States

Since 1996, the Organization for Economic Cooperation and Development (OECD) has compiled data concerning member countries' assistance to nonmember countries in strengthening national capability and capacity for environmentally sound management of pesticides and other chemicals. In the most recent compilation, 18 OECD member countries and the Commission of the European Union report 194 pesticide projects, and another 77 projects covering both pesticides and (other) chemicals.^{[4]a}

Activities frequently reported are the following:

- Risk management and risk reduction.
- Registration and classification.
- Safe use, export/import, and disposal of obsolete pesticides.
- Hazard and risk assessment.
- Laboratory testing and Good Laboratory Practice.
- Hazard and risk communication (labeling, Material Safety Data Sheets, etc.).

^aAustralia, Austria, Belgium, Canada, Denmark, Finland, France, Germany, Hungary, Japan, the Netherlands, Norway, Portugal, South Korea, Sweden, Switzerland, United Kingdom, and the United States.



Fig. 1 Middle Awash State Farm, Ethiopia, is massively oversupplied with pesticides that are now leaking into an environmentally sensitive flood plain, 1999. (Photo courtesy of Mark Davis, PAN UK.)

Without being stated as options in the questionnaire, Integrated Pest Management (IPM) or biopesticides were specifically mentioned in some ongoing projects:

- Denmark: Belarus and Bolivia.
- France: Benin, South Africa, and Tanzania.
- Germany: Egypt, Ghana, and Southeast Asia.
- Netherlands: China and Egypt.
- Norway: Vietnam, regionally in Central America, and globally through the Global IPM Facility.
- Commission of the European Union: ACP countries.^b

IOMC Partners

The Inter-Organization Programme for the Sound Management of Chemicals (IOMC) was established in 1995 to strengthen cooperation and increase coordination in the field of chemical safety. OECD and six UN agencies contribute to the work of IOMC. An attempt to quantify the extent of IOMC activities in capacity building in pesticide management is shown in Table 1.^[5]

Information Exchange Network

A new Information Exchange Network on Capacity Building for the Sound Management of Chemicals (INFOCAP) is currently (July 2003) under construction.^[6] INFOCAP is hosted by the World Health Organization (WHO) and sponsored by the

^bCountries in Africa, the Caribbean, and the Pacific region with a particular relationship to the European Union (Lomé Convention and updates); www.acpsec.org.



Fig. 2 Empty cans that have contained chlorpyrifos (Dursban) and endosulfan (Callisulfan) before being taken over the border, September 2002. (Photo courtesy of Simon Ferrigno, PAN UK.)

Intergovernmental Forum on Chemical Safety (IFCS), OECD, United Nations Institute for Training and Research (UNITAR), the Commission of the European Union, and the European Chemicals Bureau (ECB).



Fig. 3 Fruit grower spraying ethephon without any form of protection, for degreening pineapple prior to harvest in Benin, 2001. (Photo courtesy of OBEPAB.)



Fig. 4 Organic cotton producers spreading bat guano, with one of the women drinking water from an old (1989) pesticide can, Kandi, Benin, 1997. (Photo courtesy of Peter Ton.)



Fig. 5 Father and son showing off their neem-based biopesticide mixture, Benin, September 2002. (Photo courtesy of Simon Ferrigno, PAN UK.)

Table 1 IOMC involvement in capacity building for pesticide management

| Benefactor | No. of internet references |
|---|----------------------------|
| Inter-Organization Programme for the Sound Management of Chemicals (IOMC) | 934 |
| United Nations Food and Agriculture Organization (FAO) | 14,100 |
| International Labour Organization (ILO) | 4,600 |
| Organization for Economic Cooperation and Development (OECD) | 11,100 |
| United Nations Environment Programme (UNEP) | 11,300 |
| United Nations Industrial Development Organization (UNIDO) | 2,930 |
| United Nations Institute for Training and Research (UNITAR) | 811 |
| World Health Organization (WHO) | 148,000 |

Source: Ref.^[5].

Progress Indicators

The IFCS has elaborated a number of “simple indicators” to monitor progress with respect to *chemicals management capacity* and the IFCS Priorities for Action Beyond 2000:^[7] national capabilities and capacities for chemicals management; classification and labeling of chemicals; national arrangements for exchange of information on hazardous chemicals; national procedures on safety information for hazardous materials in circulation; environmentally sound and integrated strategies for pest management; obsolete stocks of pesticides and other chemicals; national systems for prevention of major industrial accidents and emergency, preparedness, and response; poison information or control centers; pollutant release and transfer register and/or emission inventories; and prevention of illegal trafficking of toxic and other dangerous chemical products.

SWEDISH CONTRIBUTIONS

Over the last few years, the Swedish International Development Cooperation Agency (Sida) has financed projects or programs for the following:

- Studies of side effects of pesticides on the fauna in Africa.
- Procurement of pesticides for countries in Africa and Asia.

⁵Implementing Company: Scanagri Sweden AB, www.scanagri.com.

- Export of organically grown products from African countries.
- Capacity building for pesticide analysis in countries in Africa and Central America.
- Disposal of obsolete pesticide stocks in Africa.
- Integrated pest management in Central America.
- Research collaboration.
- Awareness-raising and “safe use” activities through farmworkers’ unions in African countries.

Sustainable Agriculture and IPM

Sida has issued a position paper on sustainable agriculture.^[8] Principles of integrated pest management and the use of less hazardous pesticides and other related issues are covered in a draft Sida position paper on pesticides and pest management in Swedish development cooperation. Sida participates as one of 14 national lead institutions in the work of IPMEurope, a European Union network.

International Training Program

Sida provides some 60 international training programs in agriculture, environment, human rights, infrastructure, industry, public institutions, and social services.^[9] Special emphasis is placed on areas in which Sweden has a considerable level of expertise to offer. Between 1979 and 2001, some 25,000 individuals from 125 countries participated in these activities. Programs with relevance to pest or pesticide management include organic agriculture development, sustainable agriculture in an environmental perspective, hazardous waste management, occupational safety and health and development, and environmental journalism. A new program on pesticide management and pesticide risk reduction will be launched in 2004.^c

Minor Field Studies

Sida offers grants for minor field studies to Swedish students. Over 200 reports have been published, nine of which have direct bearing on pesticide management:^[10]

- Training on safe use of pesticides for farmers, Tanzania.
- IPM training, South Africa and Zambia.
- Botanical pesticides, South Africa and Zambia.
- Pesticide analysis, Vietnam.
- Managing pesticide risks, Mozambique.
- Determination of pesticide residues in water, Brazil.
- Exposure of fish to pesticides, Brazil.

- Determination of pesticides in rice, Vietnam.
- Alternative agriculture, Cuba.

Green Procurement

In a recent policy paper from Sida, partners in cooperation are requested to treat potentially hazardous pesticides and certain other chemicals in a manner that minimizes risks to human health and the environment.^[11] In all, the policy paper covers almost 150 chemical substances, most of them pesticides. Purchase of pesticides or other chemical substances covered by the Stockholm Convention on Persistent Organic Pollutants (POPs) and pesticides listed as Dirty Dozen pesticides by the Pesticides Action Network is prohibited. All remaining substances must be selected in consultation with local experts.

Purchase of pesticides classified by the World Health Organization as extremely hazardous (WHO class Ia) or highly hazardous (WHO class Ib) is also prohibited if less hazardous alternatives are available. In addition, the Prior Informed Consent principle shall be followed when importing or exporting any of the substances covered by the Rotterdam Convention.

Prevention and Disposal of Obsolete Stocks

Since 1998, Sida has contributed funds toward disposal or prevention of obsolete and unwanted pesticide stocks in Africa, particularly Ethiopia, under a project coordinated by FAO. A program to dispose of the estimated 50,000 tonnes of obsolete pesticides in Africa, the African Stockpiles Programme (ASP) has now been established under the auspices of the World Bank, the Global Environment Facility, the African Development Bank, CropLife International, the Basel Convention Secretariat, FAO, the African Union, Pesticide Action Network UK and Africa, UN Economic Commission for Africa, United Nations Environment Programme (UNEP), United Nations Industrial Development Organization (UNIDO), and the World Wide Fund for Nature.^[12] The first phase of disposal in seven countries will begin in 2003. Sida, in a possible forthcoming involvement, would put particular emphasis on prevention, capacity building, and knowledge transfer to make a sustainable impact in the recipient countries.

Multilateral Environmental Agreements

With respect to three major global chemicals conventions, Sida considers the following actions:^[13]

- Making use of existing Basel Convention regional training centers for specific training purposes,

^cImplementing Company: Scanagri Sweden AB, www.scanagri.com.

possibly in a joint chemicals approach with UNEP Chemicals.

- Facilitating ratification of the Rotterdam Convention by developing countries and, if requested, to strengthen national institutions and legislation in partner countries.
- Initiating assistance in implementation of the Stockholm Convention on POPs, particularly where excessive use of pesticides creates obstacles to export of agricultural produce.
- Assisting partner countries in overall chemical control emphasizing the responsibility of enterprises and with awareness raising as an important aspect.

THE WITTULSBERG INITIATIVE: A PILOT PROJECT FOR NORTH–SOUTH AND SOUTH–SOUTH COOPERATION

“Forging collaborations among diverse stakeholders takes patience, courage and commitment, but the pay-offs can be enormous.”^[14]

In 2000, an international group of pesticide regulators, scientists, NGOs, and trade unions produced a document providing a problem description—based on the situation in Costa Rica, Tanzania, and Vietnam—and recommendations aiming at reduced exposure to pesticides in developing countries.^[15] The group felt that donor agencies have a crucial role to play in supporting governments, civil society, and the international community to reduce pesticide hazards in developing countries. Donor agencies were recommended to:

- Support the national *problem identification* procedure, e.g., through compilation of a National Profile on pesticide and other chemical control infrastructure.
- Provide advice on strategies for *reduced exposure* to pesticides.
- Advise on and support *restrictions in availability* of pesticide products.
- Promote and support pesticide reduction through *less hazardous alternatives* and integrated pest management (IPM).
- Support establishment or strengthening of national poisoning surveillance systems and *poisons information or control centers*.
- Support implementation and monitoring of the revised International Code of Conduct on the Distribution and Use of Pesticides to supplement the Stockholm Convention on Persistent Organic Pollutants, and the Rotterdam Convention on the Prior Informed Consent Procedure for Certain

Hazardous Chemicals and Pesticides in International Trade.

- Support initiatives on *disposal* and prevention of obsolete pesticide stocks.
- Support information dissemination as a means of *feedback* to decision makers.
- Support multistakeholder cooperation in partner countries through a National Forum for pesticide risk reduction.

In 2001, the group met at Wittulsberg Mansion in Uppsala, Sweden. The group was of the opinion that Sida and other development agencies should consider, as a matter of urgency, supporting pesticide reduction through capacity development in developing countries, and extend support to relevant public-interest NGOs and trades unions.^[16] European development agencies should promote, through the projects they support, the same standards in developing countries as are acceptable in Europe. Aid should be prioritized to the urgent actions needed to achieve these ends and be a condition when supporting any scientific, technical, and research bodies. Development agencies should compile, make publicly available, and draw on successful and relevant experiences from projects they support.

CONCLUSION

Over the last few years, steps have been taken by the international community to coordinate international and bilateral development assistance in chemicals management through establishment of the IOMC and most recently—as a supplement to the UN system—formation of the Inter-governmental Forum on Chemical Safety (IFCS). A number of global chemicals conventions have been designed to solve global problems. To solve local (i.e., national) problems, a multistakeholder forum would allow national stakeholders, the UN agencies, the World Bank, the Commission of the European Union, and bilateral donors to collaboratively prioritize, make better use of available resources in a given country, and ideally avoid gaps as well as duplication of work.

REFERENCES

1. <http://www.simplelife.com/organiccotton/12GRLDR-NKNG.html>.
2. Akhabuhaya, J.L. Pesticides Use in Vegetable Production in Tanzania, Paper presented at the Pesticide Challenge—A Conference to Promote Safer Pest Management organized by the Pesticide Action Network, London, UK, November 26, 2002.

3. Saunyama, I.G.M.; Sithole, S.Z. Feasibility of Changing Regulations Towards Registration of Least Toxic Pesticides in Vegetable Production in Zimbabwe, Paper presented at the Pesticide Challenge—A Conference to Promote Safer Pest Management organized by the Pesticide Action Network, London, UK, November 26, 2002.
4. *Fourth Survey of OECD Member Country Development Assistance Activities for Capacity Building, prepared by the Organization for Economic Cooperation and Development (Paris), IFCS/FORUM-IV/4 INF*; 14 July 2003. www.who.int/ifcs/Documents/Forum/ForumIV/Meeting-docs/4INF_En.doc (accessed July 22, 2003).
5. www.google.com. Accessed July 24, 2003. Search terms: IOMC (or other benefactor) + capacity building + pesticides.
6. www.who.int/ifcs/infocap/.
7. *IFCS Simple Indicators of Progress, IFCS/FSC/02.25*; www.who.int/ifcs/Documents/Forum/ForumIV/documents/Indicator/SummaryReport.doc (accessed July 22, 2003).
8. *Sustainable Agriculture—A Summary of Sida's Experiences and Priorities*; May 1999. Position Paper, Swedish International Development Agency. http://www.sida.se/Sida/articles/3500-3599/3529/hallbart_jordbruk.pdf.
9. *International Training Programmes*; Swedish International Development Cooperation Agency. <http://www.sida.se/Sida/jsp/polopoly.jsp?d=2335>.
10. *Minor Field Studies*; Swedish University of Agricultural Sciences. <http://www-ibyr.adm.slu.se/utlandsstudier/mfs/Rapporter/nummerordning1.htm>.
11. *Sida's Policy for Green Procurement for Co-operating Partners*; Swedish International Development Cooperation Agency, September 2002. <http://www.sida.se/Sida/articles/5000-5099/5073/SidaPolGreenProc.pdf>.
12. <http://www.africastockpiles.org/>.
13. Gerremo, I. *Sida and Eight Multilateral Environmental Agreements: Their Implementation in Development Co-operation*; Swedish International Development Cooperation Agency, December 17 2002. (Memorandum).
14. *Sustainable Alternatives to Toxic Chemicals, Multi-Stakeholder Success Stories*; WWF, October 2000. http://www.worldwildlife.org/toxics/pubres/case_studies.pdf.
15. *Multi-stakeholder Collaboration for Reduced Exposure to Pesticides in Developing Countries—Recommendations to Sida with Particular Reference to Costa Rica, Tanzania, and Vietnam*; April 2000. www.pan-uk.org/briefing/SIDA_FIL/index.htm.
16. *Proceedings First Wittulberg Symposium on Pesticide Reduction in Developing Countries*; May 7–11 2001. <http://www.pan-uk.org/Internat/globinit/prdcproc.htm>.

Domestication of Agricultural Crops

Graham Scoles
Rosalind Ball

Department of Plant Sciences, University of Saskatchewan, Saskatoon,
Saskatchewan, Canada

INTRODUCTION

Plant domestication refers to the transformation of a plant that occurs in the wild to a plant that is adapted to an agricultural system—a crop plant. This process comes about through gradual genetic change over many generations of the plant. It relies on the fact that all wild plants are genetically variable. This variation is often subtle and only revealed when selection for a particular characteristic is imposed over many generations. Plant domestication, tightly linked to human civilizations, has resulted in a few hundred plant species that we now recognize as distinct from their wild relatives and which we refer to as crops. While in some cases the changes that have come about through domestication may be minor, in others, some crops have been domesticated to the point where they can no longer be perpetuated as a species except by the intervention of man.

HOW DID DOMESTICATION OCCUR?

Domestication probably started when hunter-gatherers began to deliberately manage their food supply. The first steps would have occurred with the annual harvesting of portions of the plant from an area where the plant grew naturally, the removal of unwanted species from an area, and, eventually, the deliberate planting of seed or other plant parts saved from the previous season's crop. This process would have required a relatively sedentary existence as opposed to the nomadic existence of many hunter-gatherers.

Repeated over many generations, selection would have favored plants with characteristics that probably would not have arisen in the wild, as they would have been detrimental to the plant's survival in the wild. This type of selection is referred to as unconscious selection and is best typified by the loss of natural seed or fruit dispersal mechanisms, when plants are repeatedly harvested. Harvesting favored those plants that retained their seeds or fruits. Selection was simply a result of biological forces imposed by the new agricultural environment and practices. At some point later, conscious selection was probably practiced. Plants that were more appealing were deliberately selected and

propagated as opposed to their less appealing relatives. In these early years, this would have been done without understanding the consequences. Sometime later, early farmers must have realized that such actions could lead to better crops. The final step in the selection process eventually led to the start of what we now consider modern plant breeding in the 19th century.

WHEN AND WHERE DID DOMESTICATION OCCUR?

The best evidence to date indicates that the first domestication of both crops (and animals) began around 8 to 10,000 BC in what is referred to as the Fertile Crescent, an area of mountains and foothills that form the western and northern boundaries of present-day Iraq and Iran. It is clear that domestication also occurred in the Americas, as crops were present there when Europeans arrived although settlement of the New World by humans via the Bering land bridge occurred well before agricultural developments in Eurasia. The most recent evidence dates domestication in Mexico at approximately 4 to 5000 BC. Other areas where plant domestication may have occurred independently are eastern Asia, western Africa, and South America (Peru).

Among the first people to give any thought to plant domestication was Darwin, whose observations of crops vs. their wild relatives appears to have played a significant role in the development of his theory of evolution. A Swiss botanist, Alphonse de Candolle, published a book entitled *Origin of Cultivated Plants*.^[1] In this book, the author used various sources, including botanical, archeological, historical, and linguistic evidence, to attempt to determine the areas of domestication of many crops. By 1951, the Russian scientist Vavilov^[2] had proposed up to eight centers where crop domestication most likely occurred. These were centers of crop diversity, areas that Vavilov had identified during his travels to contain an unusual level of genetic diversity. Some of these Vavilov considered to be primary centers of domestication, while others were thought to be secondary centers.

Other researchers have further elaborated on Vavilov's work and proposed additional secondary

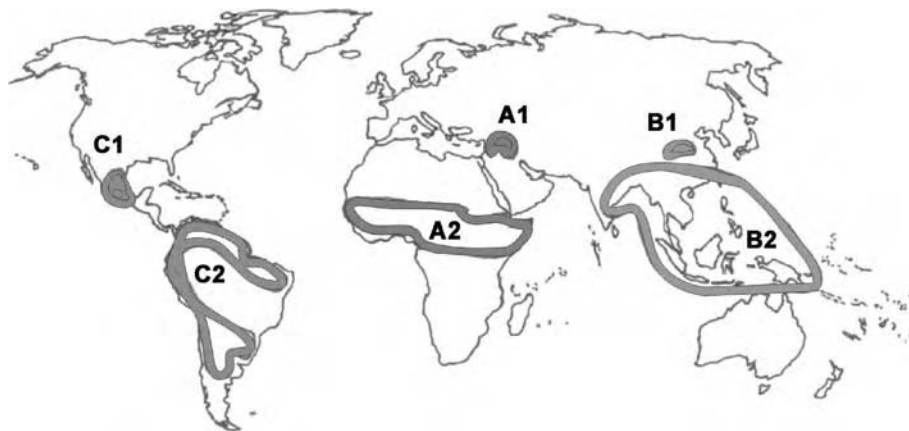


Fig. 1 Centers of plant domestication reprinted with permission from Harlan. Copyright 1971, American Association for the Advancement of Science. (From Ref.^[3].)

centers of domestication. Archeological work during the 20th century combined with radiocarbon dating has confirmed these early ideas. There is still debate as to exactly how many truly independent centers of crop domestication there were. Harlan^[3] considered that there were three main centers of domestication and three secondary centers (Fig. 1). They were: the Fertile Crescent or Near East (area A1 in Fig. 1) and its sub-Saharan Africa secondary center (A2); North China (B1) and its Southeast Asia and South Pacific secondary center (B2); and Mesoamerica (C1) and its South America secondary center (C2). Many of these regions have arid climates, lending to preservation of evidence based mainly on seeds. With more recent technological breakthroughs and sediment analysis,^[4] examination of pollen in humid lowland regions is now possible and may change or add to current ideas of where agriculture and crops originated.

CHANGES RESULTING FROM DOMESTICATION

The example of loss of dispersal mechanisms through unconscious selection mentioned above best characterizes the types of changes that came about during domestication. This is exemplified in many crops and typified by the cereals which retain their seeds well beyond maturity, whereas seeds of their wild relatives fall to the ground when mature. Presumably, plants that tended to retain their seeds would make up a larger portion of the harvest and the next year's crop. Similarly, unconscious selection at harvest probably also resulted in greater uniformity of maturity within each plant. Plants in which all seeds or fruits matured at the same time would make up a larger portion of the harvest compared to plants with asynchronous maturity. Unconscious selection may also have occurred for reduced dormancy as seed that failed to

germinate immediately would not produce plants and thus would not be included in the harvest.

Conscious selection has resulted in many types of changes. Crops from South America, such as common bean, pumpkins, cucumbers and gourds, potato, and tomato, show an amazing variety of color and array of shape among representative cultivars and closely related species (Fig. 2). Such plants have been chosen for aesthetically pleasing reasons. Another very striking difference between crops and their wild relatives is the size of the plant part that is used by man. We see this in the seeds, fruits and also the vegetative parts of crop plants including roots, stems, and leaves. Presumably, these larger plant parts were more desirable to early people although it is also possible that larger seeds and the larger seedlings they produced were favored by unconscious selection in those early fields. This is elegantly illustrated by the progression of cob size in maize (*Zea mays*) from one of its ancestors, teosinte (Fig. 3). This figure also nicely illustrates the loss of dispersal mechanisms. While the teosinte



Fig. 2 Diversity in *Capsicum chinense* (by permission of Barbara Pickersgill).



Fig. 3 Inflorescences of maize and teosinte (by permission of John Doebley).

inflorescence breaks up into small single-seeded pieces on maturity, the modern corncob is enclosed in a multilayered sheath that precludes any dispersal of the seeds on maturity.

During evolution, many plants have developed various mechanisms to provide protection from fungal, insect, and animal pests. It is very common to find these mechanisms reduced or absent in crops. These mechanisms include physical protection through structures, such as thorns and spines, and also chemical mechanisms. Many of the chemical mechanisms of protection are the result of plant-produced chemicals that not only have protective properties but also have either undesirable flavors or antidigestive properties; thus, conscious selection against them might be expected. In some cases, these compounds are still present in crop plants and may provide valuable protection. This often becomes apparent when attempts are made to reduce or remove such chemicals through plant breeding. For example, in cotton, low-gossypol cotton cultivars were bred so that the by-product seed meal could be fed to cattle; however, these cultivars sustained more insect damage in the field.

While selection has resulted in crops having desirable characteristics, it has also resulted in a tendency

toward levels of genetic uniformity that are rarely found in wild plants. The constant pressure for a harvestable and marketable product often means that commercial farmers require crop uniformity, i.e., plants achieving similar growth and development throughout the season. While this uniformity may be desirable from a number of points of view, when combined with high crop densities and large acreages, this imposes increased selection pressure on pests to evolve and attack crops. Other changes that have come about during domestication probably also favor pest attack. The reduction in natural defense mechanisms is a clear example of how domestication has had a role to play in the susceptibility of crops to pests. The increased size of various plant parts, while providing greater nutrition for humans, probably also provides more resources for pests.

Today's plant breeders spend much of their effort attempting to stay ahead of various pests as they adapt to new forms of the crop. Crop resistances to pests will eventually break down as a pest, evolves, and overcomes the crop's resistance. Plant breeders need to be aware of the changes in the pest population and take these into account in their breeding programs. In most cases, plant breeders are able to stay ahead of the pest and incorporate appropriate resistance into new cultivars. However, occasionally, this has not happened. In the 1970s, hybrid maize production in the United States was devastated by an epidemic of southern corn leaf blight.^[5] The pathogen successfully attacked plants with a particular cytoplasm, which was identical in all these hybrids. This provided a timely reminder that our domesticated plants represent an unbalanced ecosystem that requires continuous inputs in order to provide the world's food, feed, fiber, and fuel.

REFERENCES

1. de Candolle, A. *Origin of Cultivated Plants*, 2nd Ed.; translated from the 1886 edition; Hafner: New York, 1959; 468 pp.
2. Vavilov, N.I. The origin, variation, immunity and breeding of cultivated plants. *Chron. Bot.* **1951**, *13*, 1–366.
3. Harlan, J.R. Agricultural origins: centers and noncenters. *Science* **1971**, *174*, 468–474.
4. Pope, K.O.; Pohl, E.D.; Jones, J.G.; Lentz, D.L.; von Nagy, C.; Vega, F.J.; Quitmyer, I.R. Origin and environmental setting of ancient agriculture in the lowlands of Mesoamerica. *Science* **2001**, *292*, 1370–1373.
5. Tatum, L.A. The southern corn leaf blight epidemic. *Science* **1971**, *171*, 1113–1116.

Energy Cost and Use in Pesticide Production

Diego O. Ferraro

IFEVA, Departamento de Recursos Naturales y Ambiente, Universidad de Buenos Aires—CONICET, Buenos Aires, Argentina

INTRODUCTION

Modern agricultural systems are artificially manipulated to produce food and fiber. Current agricultural practices reduce the plant component of these systems to one or two dominant species. The persistence of this unnatural ecosystem design involves the use of complementary sources of energy to control the growth and development of undesired community components (e.g., pests and weeds). This additional energy requirement substantially reduces the energy efficiency of agroecosystems compared with natural systems. Considerations such as the energy used in pesticide and fertilizer production are often overlooked in assessing the energy efficiency of agriculture. Pesticides (i.e., herbicides, insecticides, and fungicides) are important inputs to control insect, weed, and plant pathogen populations in agricultural fields. In addition to pesticide toxic and pollution risks, pesticides also involve an important component of energy utilization in the manufacturing process.

ENERGY USE IN AGRICULTURE

Since about 1950, when agriculture became extensively mechanized in the western part of the world, pesticide use also started. Because of these technology changes, agricultural yields have been growing and productive land area has been augmented.^[1] However, these benefits have turned modern agricultural models into high-energy-dependent systems. Therefore the extent of the use of farm machinery and pesticides depends on fossil fuel availability as the most common source of energy. Particularly, pesticide manufacturing involves the extensive use of energy during production. Because an agricultural system comprises natural processes that are ruled by thermodynamics, energy utilization has to be analyzed with the aim of assessing energetic efficiency in the management of natural resources.

Energy input is the amount of direct and indirect energy required to produce a given resource. The composition of the energy use in agricultural production can be divided in direct and indirect energy inputs.

Direct energy refers to the fuel burned at the site of production, such as a chemical plant. Indirect energy refers to the fuel burned outside the chemical plant. Energy commodities (direct energy inputs) are highly visible energy requirements for agriculture. In contrast, indirect energy requirements are less visible, or may be hidden. While about one-third of the energy consumed in the farm is for direct use, nearly two-thirds of the energy is consumed indirectly.^[2] These indirect energy costs can be very important and can influence technologies employed in agriculture and society.

The manufacturing cost of pesticides, farm equipment, and fertilizers comprises around 90% of the energy used in agriculture. The energy costs of fertilizer and farm machinery in agricultural production account for about two-thirds of the energy. Pesticide use encompasses 2–4% of the total energy used in the crop production process.^[3,4] The average energy input in the production, transportation, and application of pesticides was 6.6% of the total energy used in the production of fertilizers in the United States in 1980.^[5,6] Although pesticides may represent only a small portion of the total energy invested on agriculture, based on per unit weight of input, more energy is involved in the production and application of pesticides than any other input agriculture. On average, the production of pesticides takes four to five times more energy per kilogram than nitrogen fertilizer production.^[5]

ENERGY USE IN PESTICIDE MANUFACTURING PROCESS

The manufacture of pesticides is a highly complex process, resulting in high-energy inputs per kilogram produced. Physical, chemical, and thermodynamic characteristics of the manufacturing process determine the energy cost of a pesticide. Most pesticides are derived from ethylene and propylene, which are obtained by catalytic cracking of crude petroleum oils, or from methane from natural gas. The total energy cost is the sum of the energy sequestered in the material itself and that required to apply it to the crops. Some pesticides are more energy-intensive than others (Table 1). The energy requirements of

Table 1 Energy per Kilogram (E_w), average of recommended field rate (FR), energy per Hectare (E_{ha}), and Toxicity Class (TC) of various pesticides

| Pesticide | E_w^a (Mcal/kg AI) | FR (kg AI/ha) | E_{ha} (Mcal/ha) | TC ^b |
|---------------------|----------------------|---------------|--------------------|-----------------|
| <i>Herbicides</i> | | | | |
| MCPA | 30.9 | 2.00 | 61.8 | Low |
| 2,4-D | 20.2 | 0.50 | 10.1 | Low |
| Dicamba | 70.2 | 0.27 | 18.954 | Low |
| Chloramben | 40.4 | 3.60 | 145.44 | Low |
| Fluazifop-butyl | 123.2 | 1.20 | 147.84 | Low |
| Propanil | 52.3 | 4.00 | 209.2 | Low |
| Alachlor | 66.1 | 4.00 | 264.4 | Low |
| Propachlor | 69.0 | 5.00 | 345 | Low |
| Chlorsulfuron | 86.8 | 0.04 | 3.472 | Low |
| Butylate | 33.5 | 5.00 | 167.5 | Very low |
| Diuron | 64.2 | 1.00 | 64.2 | Low |
| Fluometuron | 84.4 | 2.28 | 192.432 | Very low |
| Atrazine | 45.2 | 2.50 | 113 | Low |
| Trifluralin | 35.7 | 2.00 | 71.4 | Low |
| Diquat | 95.2 | 2.00 | 190.4 | Moderate |
| Paraquat | 109.4 | 1.60 | 175.04 | Moderate |
| Glyphosate | 108.0 | 2.20 | 237.6 | Very low |
| Linuron | 69.0 | 2.50 | 172.5 | Low |
| Cyanazine | 47.8 | 1.80 | 86.04 | Moderate |
| Bentazon | 103.2 | 0.80 | 82.56 | Low |
| EPTC | 38.1 | 5.00 | 190.5 | Low |
| Metolachlor | 65.7 | 1.50 | 98.55 | Low |
| <i>Fungicides</i> | | | | |
| Benomyl | 94.5 | 0.70 | 66.15 | Very low |
| Captan | 27.3 | 2.50 | 68.25 | Very low |
| Ferbam | 15.2 | 3.20 | 48.64 | Low |
| Maneb | 23.5 | 1.30 | 30.55 | Very low |
| Sulfur | 26.6 | 12.00 | 319.2 | Very low |
| <i>Insecticides</i> | | | | |
| Carbaryl | 36.4 | 2.00 | 72.8 | Low |
| Carbofuran | 108.1 | 1.30 | 140.53 | High |
| Cypermethrin | 138.1 | 0.13 | 17.953 | Low |
| Lindane | 13.8 | 7.50 | 103.5 | Moderate |
| Malathion | 54.5 | 1.80 | 98.1 | Very low |
| Methyl parathion | 38.1 | 1.00 | 38.1 | High |
| Parathion | 32.8 | 0.60 | 19.68 | High |
| Methoxychlor | 16.7 | 1.14 | 19.038 | Very low |

^aSource: Refs.^[4-6].

^bToxicity classes are categorized according to acute oral toxicity for rats, measured by lethal dose 50 (LD₅₀ in mg/kg): 1-50 = high; 50-500 = moderate; 500-5000 = low; and over 5000 = very low.

insecticide compounds, with an average value of 51.01 Mcal/kg active ingredient (AI), are slightly less than for herbicides (average = 62.09 Mcal/kg AI) whereas fungicides (37.42 Mcal/kg AI) appear to be the most economical group (Table 1). The energy inputs for each pesticide range from 15.2 Mcal/kg AI for ferbam to 138.1 Mcal/kg AI for cypermethrin. These inputs vary according to the energy contained in hydrocarbon feedstocks used and the amount of heat and electricity used in the manufacturing process.

After the AI has been manufactured, it is combined with other materials into a formulation, such as oil and wettable powder, that is then packaged and shipped to the farm. The final energy value of pesticide use also comprises formulation, packaging, and transportation. Energy costs for formulation, packaging, and transport for different formulation techniques are shown in Table 2. These energy components of pesticide production average about a third of the total energy inputs,^[3] but the final proportion depends on the type of formulation. Miscible oil contains hydrocarbon-based

Table 2 Energy inputs for different kinds of formulations (Mcal/kg AI)

| | Formulation | Packaging | Transportation | Total |
|-----------------|-------------|-----------|----------------|-------|
| Miscible oil | 33.2 | 8.5 | 1.1 | 42.8 |
| Wettable powder | 2.5 | 2.6 | 0.7 | 3.6 |
| Granules/dust | 3.6 | 20.0 | 6.7 | 30.4 |

Source: Ref.^[4].

solvents, thus additional energy is required to make pesticides into this formulation. The least energy-intensive way of supplying a pesticide appears to be a wettable powder formulation because it does not require any hydrocarbon-based solvents.

ON-FARM APPROACH

To assess the total energy consumption related to pesticide use, it is crucial to examine the on-farm extent of pesticide utilization. Different crop production systems may vary in the total required energy per hectare. When considering the energy inputs for producing a hectare of corn, pesticides represent about 8–15% of the total energy inputs; in soybean crops, they represent 24% of the total energy inputs and, in wheat, they represent only 3%.^[2,7,8] These differences might be explained by the qualitative and quantitative diversities of pests and pesticides used on each crop. Table 1 shows the energy used per unit area of various pesticides applied at the recommended dose. Final energy per unit area appears to be highly modified by the dose applied. Total energy per hectare ranges from 3.47 Mcal/ha for chlorsulfuron to 345 Mcal/ha for propachlor. However, this difference is not evident when only the manufacturing and application costs are considered. Furthermore, the pesticide that shows the highest energy per area of application (propachlor) is produced using 20% less energy per unit weight than chlorsulfuron, which is the analyzed pesticide with the lowest energy per area value (see Table 1 for different examples of the same pattern). Although the trend in pesticide manufacturing is toward the production of pesticides that are more energy-intensive per unit, the application is very low in rates per hectare. This analysis highlights the fact that energy inputs in pesticides might be considered as a part of the total agricultural system.

A complete framework to analyze the energy costs in pesticide use should take into consideration the environmental and social costs incurred beyond manufacturing and application in agriculture. Such extra costs include farm worker medical expenses, monitoring of food for residues, drift of pesticides onto neighboring farms or urban areas, and water quality.

However, one of the main extra costs is associated with the effects of pesticides on wildlife. Pesticides show a high heterogeneity in their toxicity effect on the biotic components of agroecosystems (Table 1). Herbicides appear to be less toxic to wildlife. This aspect is very important because the pesticide effects on the ecosystem services are often ignored. The density of pollinators and natural enemies of pests and the integrity of decomposer soil webs are among the ecosystem properties mainly affected by pesticide use.^[9] These natural biological agents save farmers billions of dollars annually by protecting crops and reducing the need for chemical control, resulting in energy savings and reduced costs.

CONCLUSION

To summarize, pesticides play a major role in modern agricultural systems and imply significant quantities of fossil energy. This energy is directly utilized in the manufacturing process and indirectly utilized in formulation, packaging, and transportation. Although there is a great variability among pesticides, herbicides appear to be slightly less energy-expensive than insecticides, and fungicides appear to be the most economical group. However, the energy inputs analysis should be matched with the assessment of the environmental and public health effects of pesticides. The heavy use of pesticides has significant economic (and energetic) consequences.

REFERENCES

1. Kidd, C.V. Food production. In *Integrated Resource Management: Agroforestry for Development*; Kidd, C.V., Pimentel, D., Eds.; Academic Press, Inc.: Toronto, 1992; 3–28.
2. Pimentel, D. Energy inputs in production agriculture. In *Energy in World Agriculture*; Fluck, R.C., Ed.; Elsevier: Amsterdam, 1992; 13–29.
3. Refsgaard, K.; Halberg, N.; Kristensen, E.S. Energy utilization in crop and dairy production in organic and conventional livestock production systems. *Agric. Syst.* **1998**, *57*, 599–630.

4. Pimentel, D. *Handbook of Energy Utilization in Agriculture*; CRC Press: Boca Raton, FL, USA, 1980.
5. Green, M.B. Energy in pesticide manufacture, distribution and use. In *Energy in Plant Nutrition and Pest Control*; Hesel, Z.R., Ed.; Energy in World Agriculture; Elsevier: Amsterdam, 1987; Vol. 2, 165–177.
6. Hesel, Z.R. Energy and alternatives for fertilizer and pesticide use. In *Energy in Farm Production*; Fluck, R.C., Ed.; Energy in World Agriculture; Elsevier: New York, 1992; Vol. 6, 177–201.
7. Swanton, C.J.; Murphy, S.D.; Hume, D.J.; Clements, D.R. Recent improvements in the energy efficiency of agriculture: case studies from Ontario, Canada. *Agric. Syst.* **1996**, *52*, 399–418.
8. Clements, D.R.; Weise, S.F.; Brown, R.; Stonehouse, D.P.; Hume, D.J.; Swanton, C.J. Energy analysis of tillage and herbicide inputs in alternative weed management systems. *Agric. Ecosyst. Environ.* **1995**, *52*, 119–128.
9. Daily, G.C.; Alexander, S.; Ehrlich, P.R.; Goulder, L.; Lubchenco, J.; Matson, P.A.; Mooney, H.A.; Postel, S.; Schneider, S.H.; Tilman, D.; Woodwell, G.M. Ecosystem services: benefits supplied to human societies by natural ecosystems. *Issues Ecol.* **1997**, *2*, 1–16.

Energy in Pesticide Production and Use

Zane R. Helsel

Department of Extension Specialists, Cook College, Rutgers University,
New Brunswick, New Jersey, U.S.A.

INTRODUCTION

The use of pesticides to control weeds, insects, diseases, and other pests has been a significant force in industrialized agriculture allowing for the cultivating of large areas by a small number of laborers. With the use of pesticides have come some challenges of maintaining the health of the environment and its inhabitant's; the interruption of natural biological cycles; and the use of fossil fuels for its manufacture, distribution, and application. The advent of safer and more energy-efficient pest control measures have negated much of these challenges. This article highlights the energies involved in pesticide production and use and the effects of various management alternatives on energy for pest control.

OVERALL PESTICIDE AND ENERGY USE

Pesticide use worldwide totaled 2432 million kg of active ingredients (AIs) in 2000 (Table 1) with consumption in the U.S.A. totaling nearly one-fourth of that.^[1] Herbicides comprised over one-third of all pesticide use worldwide, followed by insecticides, representing one-fourth. In the U.S.A., herbicides represented nearly half the amount of pesticides used and was dominated by the use of glyphosate and atrazine. The use of pesticides also vary by commodity.^[2] In the U.S.A., the greatest amount on a per hectare basis was used by fruit and vegetables, but by virtue of their vast hectarage, the feed and food grain crops dominated the total use overall. Forages and pastures, in general, utilized the least per area and in total.

While significant amounts of pesticides are used worldwide, the average energy consumed in pesticide use represents less than 15% of the overall total energy used in agriculture.^[3] Fertilizer (nitrogen in particular); irrigation; and grain drying, followed by direct fuel for field operations, represent the greatest amounts of energy in agriculture production.^[4] Nevertheless, pesticide manufacture can require 2–5 times as much energy per kilogram than nitrogen fertilizer manufacture. For greater detail on pesticide use and comparative energies in agriculture, the reader is directed to Pimentel,^[3] Stout,^[4] and Helsel.^[5]

ENERGY INVESTED IN PESTICIDE MANUFACTURE

Energy in pesticides varies by the type of the chemical and the resources to manufacture them. Many pesticides are derived from petroleum chemicals, mainly ethylene, propylene, and methane. Electricity, natural gas, steam, and/or other petroleum sources are also used in manufacturing, for such processes as heating, distillation, stirring, and drying. Secondary and tertiary inputs of energy also occur in the construction and maintenance of the manufacturing plant and equipment, import of raw materials, export of waste, and the many energies involved in human operations. A more detailed discussion of these energies, calculations thereof, and cost-benefit analyses can be found in a treatise by Green.^[6]

Table 2 contains a summary of estimated energy requirements, on a per kilogram of AI basis, for the manufacture of pesticides and on a per hectare basis for typical use. While data is presented for many of the products used in the greatest amounts in the U.S.A. and the world, little information is available on newer materials because of proprietary rights of manufacture and processes. Also not included are the many new biopesticides coming on the market. Biopesticides use few hydrocarbon fuels in their chemical makeup, but still consume energy for the overall manufacture and use. It should be cautioned that the listed values (Table 2) of older off-patent chemicals may be off by a factor of $\pm 10\%$ and the newer patented or even off-patent products may vary from true values by up to 50%. The trend in manufacturing has been to find more efficient methods of production so newer plants manufacturing older chemical may have lower actual energy consumption than originally calculated.

Pesticides differ not only in energy of manufacture but, because of different use rates, also vary in energy use per hectare (Table 2). Listed are typical use rates for one or more major crops during a growing season. The reader is cautioned that rates vary based on crop use, edaphic conditions, method of application, and pest problems. Also, in some cases, pesticides are applied multiple times to the same crop in the same growing season. With the major onset of genetically

Table 1 World and US pesticide use, 2000^a

| Type | World market | | US market | | USA % of world market |
|-------------------------|---------------------------------|-----|--------------------|-----|-----------------------|
| | Million kg of A.I. ^b | % | Million kg of A.I. | % | |
| Herbicides ^c | 884 | 36 | 246 | 44 | 28 |
| Insecticides | 616 | 25 | 55 | 10 | 9 |
| Fungicides | 235 | 10 | 34 | 6 | 14 |
| Other ^d | 698 | 29 | 225 | 40 | 32 |
| Total | 2432 | 100 | 561 | 100 | 23 |

^a(Adapted from Ref.^[1].)

^bA.I. = active ingredient.

^c“Herbicides” includes herbicides and plant growth regulators.

^d“Other” includes nematicides, fumigants, rodenticides, molluscides, aquatic and fish/bird pesticides, other miscellaneous pesticides, plus other chemicals used as pesticides (e.g., sulfur and petroleum oil).

engineered glyphosate-resistant crops, this chemical has vaulted into the number one used herbicide in the U.S.A. in the year 2000.^[1] On a per kilogram of AI, glyphosate requires more than twice the energy for manufacture (454 MJ/kg) than that of atrazine (190 MJ/kg), and more than one and a half times that of metolachlor (276 MG/kg), the two herbicides that glyphosate is replacing in corn production. However, both atrazine and metolachlor were and are used together and are needed to obtain nearly similar weed control as that provided by glyphosate alone. This, coupled with the fact that glyphosate is used at a lower rate per hectare, results in a calculated use of energy per hectare for glyphosate of 25% less than the two herbicides it is replacing and is considered more environmentally benign.

Another comparison illustrates the continuing trend of significantly lower rates of AI per hectare that result from the use of the more concentrated energy-intensive pesticides. Fluzifop-butyl represents one of the first patented of these newer age, lower use rate chemicals. While its invested energy of manufacture is nearly two times that of metolachlor on a per kilogram of AI basis, on a comparable rate of application to control similar grassy weeds, the energy for the manufacture of fluzifop-butyl is only about one-third that of metolachlor on a per hectare basis.

ENERGY IN FORMULATION, PACKAGING, TRANSPORT, AND APPLICATION

Although energy in manufacture is a significantly large part of the overall energy invested in pesticide use, energies for formulation, packaging, and transportation can also add measurable amounts to the total energy expended to deliver useable pesticides to the farm gate. These energies can vary greatly, particularly with today's agriculture having many formulations and packaging options. Green^[6] suggests that emulsifiable

oil-based pesticides may require about 20 MJ/kg, wettable powders up to 30 MJ/kg, granules 10 MG/kg, and microgranules 20 MJ/kg for formulation. Packaging is estimated to require about 2 MJ/kg and transportation about 1 MJ/kg. These energies can also vary widely by pesticide, and as would be postulated, products like fluzifop-butyl, which are concentrated and applied at very low rates per hectare, have very little energy expended on a per hectare basis for formulation, packaging, and distribution.

Once pesticides reach the farm, energy is expended in application to the crop.^[4] Before some pesticides are applied, “crop oils” may be added to the tank mixture for enhanced efficacy. Typical rates could be 2–4 L/ha. For application, typical broadcast operations by a tractor and tank sprayer require 1–2 L/ha of fuel. If combined as part of the field tillage operations, the extra energy expended is insignificant. Some specialized equipment, such as air blast sprayers in orchards, can consume significantly more fuel (6 L/ha). Aircraft spraying application may also consume more than ground applications, if fields are small and turning is frequent. New low-volume application technology can lower application energies, particularly by reducing refills and transport weight.

ALTERNATIVE PEST CONTROL PRACTICES TO LOWER ENERGY USE

Although pesticides represent less than 15% of the energy used in the production of many crops, and energy use per hectare is decreasing, it is still important to consider alternatives to reduce energy expenditures. An often-queried consideration is to replace pesticide use with non-chemical pest control measures. Because herbicides are such a significantly large component of the pesticide market, some have considered mechanical cultivation as a way to reduce pesticide use and energy consumption. A typical comparison in US soybean

Table 2 Estimated manufacturing energy inputs for various pesticides (MJ/kg), typical application rates (kg/ha) and energy per unit area of use (MJ/ha) on an AI basis^a

| Pesticide | Application rate | | MJ/ha |
|---------------------|------------------|---------|-------|
| | MJ/kg | (kg/ha) | |
| <i>Herbicides</i> | | | |
| 2,4-D | 85 | 0.50 | 43 |
| Alachlor | 278 | 2.75 | 765 |
| Atrazine | 190 | 1.75 | 333 |
| Bentazon | 434 | 1.00 | 434 |
| Butylate | 145 | 3.50 | 254 |
| Chlorsulfuron | 365 | 0.025 | 9 |
| Dicamba | 295 | 1.00 | 295 |
| Dinoseb | 80 | 2.00 | 160 |
| Diquat | 400 | 0.50 | 200 |
| Diuron | 270 | 2.25 | 608 |
| EPTC | 160 | 4.00 | 640 |
| Fluazifop-butyl | 518 | 0.25 | 130 |
| Fluometuron | 354 | 1.50 | 533 |
| Glyphosate | 454 | 1.25 | 567 |
| Linuron | 290 | 1.00 | 290 |
| MCPA | 130 | 0.55 | 72 |
| Metolachlor | 276 | 1.50 | 345 |
| Paraquat | 450 | 0.55 | 414 |
| Propachlor | 290 | 3.00 | 870 |
| Propinil | 220 | 4.00 | 880 |
| Trifluralin | 150 | 1.00 | 150 |
| <i>Fungicides</i> | | | |
| Benomyl | 397 | 0.55 | 218 |
| Captan | 115 | 3.35 | 385 |
| Ferbam | 61 | 8.00 | 488 |
| Maneb | 99 | 4.00 | 396 |
| <i>Insecticides</i> | | | |
| Carbaryl | 153 | 1.75 | 268 |
| Carbofuran | 454 | 2.00 | 908 |
| Cypermethrin | 580 | 0.25 | 145 |
| Malathion | 229 | 1.25 | 286 |
| Methoxychlor | 70 | 0.50 | 35 |
| Methyl Parathion | 160 | 1.00 | 160 |
| Phorate | 209 | 2.50 | 523 |

^a(Adapted from Ref.^[6].)

production today would be the use of approximately 1.25 kg/ha of glyphosate applied in one postemergence operation requiring a total of slightly more than 600 MJ/ha of energy from manufacture to application. A traditional mechanical approach would utilize at least one rotary hoeing (80 MJ/ha) and two standard cultivations (310 MJ/ha) for a total of 490 MJ/ha direct diesel fuel equivalent, plus an estimated additional 100 MJ/ha for indirect energies associated with fuel acquisition and processing and farm equipment manufacture. While the estimated energy totals for both methods of weed control are similar, the mechanical alternative could likely result in reduced weed control particularly under wet conditions (as might also result with some herbicides under dry conditions), and therefore lower soybean yields.

With the advent of genetic engineering and the incorporation of insect resistance into the germplasm of various crops, the need for energy-intensive insecticides will potentially be greatly reduced in corn, cotton, and several other major crops. Use of biopesticides can also reduce energy use if volumes and methods of application are not excessive. Good overall pest management provides less dramatic, but nonetheless, significant opportunities to reduce pesticide use and thus energy use in crop production. The use of integrated pest management (IPM), which involves scouting for pests, and the determination of economic thresholds of pests can reduce the calendarization and routine of frequent spraying of preventative pesticides. In heavy use pesticide situations, such as for fruits and vegetables, 50% or more reduction in pesticide use can sometimes be realized from using IPM.

Other good crop stewardship such as adequate fertility, crop rotations, cover crops, proper plant spacing, and optimal planting dates can also often reduce the amount of pesticide needed per hectare. Use of low-volume/low-rate technologies, and substitution of lower energy materials or non-petroleum based pesticides can also lower overall energy expended in crop production.

CONCLUSIONS

Manufacture of pesticides is a fossil fuel energy-intensive process. However, pesticide energy used in agriculture averages less than 15% of the total energy invested in agriculture overall. New concentrated pesticide products and formulations, while energy intensive in manufacture, are used in ultralow amounts, thus greatly reducing energy use per hectare. Because pest control is important both in yield and quality of crops, it is of utmost importance to first choose the best pesticide(s) and/or other control methods, then evaluate methods to reduce total amounts of energy in the various processes. These practices will often provide significant reductions in pesticide energies per unit energy of crop production compared to selecting a pesticide based solely on low fossil fuel energy in manufacture that may sacrifice pest control.

ARTICLES OF FURTHER INTEREST

- Adjuvants and Carriers*, p. 1.
- Aerial Application*, p. 7.
- Airblast Sprayers*, p. 11.
- Biopesticides*, p. 85.
- Biotechnology*, p. 1.
- Controlled Droplet Application*, p. 148.

Genetic Improvement of Biocontrol Agents, p. 329.
Oils, p. 1.
Pest Management in Organic Farming, p. 580.
Transgenic Crops (Annuals), p. 846.
Transgenic Crops (Perennials), p. 850.

REFERENCES

1. Kiely, T.; Donaldson, D.; Grube, A. *Pesticide Industry Sales and Usage 2000 and 2001 Market Estimates*; USEPA: Washington, DC, 2004.
2. <http://www.usda.gov/nass/pubs/estindx1.htm#agchem> (accessed May 2005).
3. Pimentel, D. Energy inputs in production agriculture. In *Energy in Farm Production*; Fluck, R.C., Ed.; Energy in World Agriculture; Elsevier: Amsterdam, 1992; Vol. 6, 13–29.
4. Stout, B.A. *Energy Use and Management in Agriculture*; Breton Publishers: N. Scituate, Massachusetts, U.S.A., 1984.
5. Helsel, Z.R. Energy and alternatives for fertilizer and pesticide use. In *Energy in Farm Production*; Fluck, R.C., Ed.; Energy in World Agriculture; Elsevier: New York, 1992; Vol. 6, 177–201.
6. Green, M.B. Energy in pesticide manufacture, distribution, and use. In *Energy in Plant Nutrition and Pest Control*; Helsel, Z.R., Ed.; Energy in World Agriculture; Elsevier: Amsterdam, 1987; Vol. 2, 165–177.

Enhanced Microbial Degradation of Pesticides

Todd A. Anderson

Institute of Environmental and Human Health, Texas Tech University, Lubbock, Texas, U.S.A.

Joel R. Coats

Department of Entomology, Iowa State University, Ames, Iowa, U.S.A.

INTRODUCTION

A change in the persistence of some pesticides can occur with repeated use, resulting in a lack of control of a particular crop pest. This phenomenon, termed enhanced microbial degradation or accelerated pesticide degradation, is the result of soil microorganisms becoming adapted or acclimated to a particular pesticide or class of pesticides. Depending on one's perspective, the phenomenon of enhanced microbial degradation of pesticides can be advantageous or detrimental. Those in the agriculture industry depend on some pesticide persistence to achieve effective crop protection. However, environmentalists often worry that persistent pesticides are more likely to have a negative impact on nontarget organisms. Ultimately, the balance between these issues is controlled by soil microorganisms and their metabolic capabilities, allowing the use of crop protection chemicals in food and feed production while still reducing actual or possible hazards to organisms and maintaining soil and environmental quality.

MICROBIAL GROWTH AND ADAPTATION

Microbial populations in pure culture exhibit a growth curve with four recognized phases: lag, log, stationary, and death. The lag phase is the period in which no growth occurs. The length of this phase represents enzyme induction, or the time required for production of enzymes necessary to utilize a new substrate. The log phase is characterized by exponential growth of the microbial population as the new substrate is utilized. The stationary phase is reached when population growth slows or halts because of nutrient limitation. The final phase, death, is characterized by a decline in the microbial population. It is believed that microorganisms in soil operate under a different, although similar, growth curve as microorganisms grown in laboratory culture.^[1] Soil microorganisms are largely dormant, but rapidly increase their growth in response to favorable conditions such as the presence of a new substrate. In addition, soil is characterized by a mixed

culture (several populations) of microorganisms that can interact, rather than a pure culture (single population) of microorganisms.

Microbial substrate in soil is largely provided by organic constituents from decaying plant and animal tissues. Plants also exude materials into the root zone. These compounds include lipids, proteins, and lignin, in addition to unidentifiable macromolecules found in soil (humus). As might be expected, soil microorganisms can utilize these structurally diverse materials for growth. In fact, the microbial community in soil is capable of degrading virtually any organic compound that is added to soil.^[2]

Soil microorganisms have an incredible capability to adapt to new carbon sources entering the soil environment. Through mutation and enzyme induction, microorganisms can develop the ability to degrade substrates that previously could not be utilized, giving adapted microorganisms an evolutionary advantage. The primary mechanism for this adaptation is the production of new enzymes through the possession and utilization of plasmid DNA. Plasmids are extrachromosomal DNA that exist independently of the chromosome. Plasmids, in general, and degradative plasmids, in particular, are important because they can be transferred to other bacteria of the same or different species.^[2] The plasmid confers on the new host the capability to metabolize the substrate that it previously was unable to utilize.

MECHANISMS OF ENHANCED PESTICIDE BIODEGRADATION

Because the degradation of organics in soil is a largely biotic phenomenon dependent on favorable conditions for microbial growth and activity, enhanced microbial degradation of pesticides is the result of soil microorganisms adapting to a new source of carbon and energy. Increases in microbial biomass following the first addition of a pesticide can result in rapid degradation of the pesticide following subsequent pesticide additions. Alternatively, greater activity per cell^[3] through the increased production of degradative

enzymes can lead to decreased persistence of pesticides. Regardless of the mechanisms involved, soil microorganisms with the ability to quickly utilize new substrates will have a selective advantage over those microorganisms that cannot utilize these new substrates. Ultimately, the advantaged microorganisms are more likely to multiply and reach a higher population density in soil.

CLASSIC EXAMPLES OF ENHANCED PESTICIDE BIODEGRADATION

The phenomenon of enhanced microbial degradation of pesticides was first observed in the laboratory more than 50 years ago. Ironically, the observation was made using one of the first synthetic pesticides ever developed: 2,4-dichlorophenoxyacetic acid (2,4-D). Initially, soil columns to which solutions of 2,4-D were added showed a defined lag period in which little 2,4-D was degraded, followed by a period of rapid 2,4-D degradation.^[4] Subsequent doses of 2,4-D to the previously treated soil column were degraded without a lag phase, indicating the acclimation of some soil microorganisms to 2,4-D. Follow-up studies also revealed that soils pretreated with 2,4-D could degrade the compound much faster than untreated soils.

In the field, the phenomenon of enhanced microbial degradation of pesticides was not reported until the mid-1970s. Carbofuran, an insecticide used in corn rootworm control, began to fail in some situations where it had significant historical use.^[5] In addition, the carbamothioate herbicide, S-ethyl dipropylcarbamothioate (EPTC), no longer provided effective control of weeds in fields with historic EPTC use.^[6] These pesticide failures were ultimately linked to rapid microbial degradation brought about by the application of the particular pesticide for two or more consecutive years to the same field.

POSSIBLE APPROACHES TO REDUCE ENHANCED PESTICIDE BIODEGRADATION

Several proposed strategies for preventing or reducing the impact of enhanced microbial degradation of pesticides have been evaluated. These strategies include

attempts to avoid the development of enhanced pesticide degradation through the use of pesticide rotations, and attempts to circumvent enhanced pesticide degradation after it has developed through the use of pesticide application timing. In developing strategies for the management of enhanced microbial degradation of pesticides, it must be recognized that microbial adaptation is a natural process that is likely to be difficult to eliminate. If chemical control is to remain dominant in pest management, this concept must be recognized and accepted.

CONCLUSION

Microbial adaptation and acclimation are natural processes. Enhanced microbial degradation of pesticides is the result of soil microorganisms becoming adapted or acclimated to a particular pesticide or class of pesticides with repeated use. The mechanisms involved in the adaptation or acclimation usually involve an increase in the numbers of microorganisms (biomass) or an increase in the activity of those microorganisms. Regardless of the mechanism, the end result is a decrease in pesticide persistence. This effect may compromise the protection of crops from weed, insect, and fungal pests.

REFERENCES

1. Racke, K.D. Pesticides in the soil microbial ecosystem. In *Enhanced Biodegradation of Pesticides in the Environment*; Racke, K.D., Coats, J.R., Eds.; American Chemical Society: Washington, DC, 1990; 1–12.
2. Alexander, M. *Biodegradation and Bioremediation*; Academic Press: San Diego, CA, 1994; 302 pp.
3. Moorman, T.B. Populations of EPTC-degrading microorganisms in soils with accelerated rates of EPTC degradation. *Weed Sci.* **1988**, *36*, 96–101.
4. Audus, L.J. The biological detoxication of 2,4-dichlorophenoxyacetic acid in soil. *Plant Soil* **1949**, *2*, 31–36.
5. Felsot, A.S.; Maddox, J.V. Enhanced microbial degradation of carbofuran in soils with histories of furadan use. *Bull. Environ. Contam. Toxicol.* **1981**, *26*, 781–788.
6. Roeth, F.W.; Wilson, R.G.; Martin, A.R.; Shea, P.J. Enhanced soil degradation of butylate and EPTC in Nebraska fields. *Weed Technol.* **1989**, *3*, 24–29.

Equipment for Ground Applications to Adult Mosquitoes

Jane A.S. Barber

Mosquito Adulticide Section, College of Engineering Science Technology and Agriculture,
Florida A&M University, Panama City, Florida, U.S.A.

INTRODUCTION

This chapter is an introduction to the equipment used to control adult mosquitoes from the ground. Chemical control is a substantial component of all mosquito control programs, with specialized equipment used for pesticide application. The efficacy of application equipment is important because no pesticide can be more effective than the efficacy inherent in the methods used for its delivery.

BACKGROUND

The most recognized methods in the U.S.A. for ground applications of adulticides are truck mounted space sprayer applications.

- Ultra low volume (ULV), sometimes referred to as cold fogging machines
- Thermal fogging machines categorized as low volume (LV)

Small handheld versions of these machines are available for treating areas inaccessible to vehicles.

In both thermal fogging and ULV applications, aerosols are employed to produce a space spray that will drift through the target zone. The aerosol persists for an appreciable length of time at suitable droplet densities to impact upon the target—the flying mosquito; chemical concentrate is atomized into fog (drops $<50\ \mu\text{m}$), and the insecticide fog is only considered effective while the droplets remain airborne. Hence, a space spray does not have any residual effect.

Where a more long-term effect is required, another method—residual spraying—is employed for controlling adult mosquitoes. In this case, the insect is required to land on a surface deposit of the insecticide to pick up a toxic dose. Residual sprays are often referred to as barrier or surface spraying.

- A barrier spray is applied to prevent adult mosquitoes moving into an area such as a stadium, often achieved with a modified vehicle-mounted hydraulic sprayer.

- A surface spray is used to kill and exclude adults from a harborage area or resting site, often around the home, and so a handheld device is employed such as the micron ulva knapsack mist blowers or compression sprayers.

Most residual spraying occurs in areas of severe disease prevalence described by the World Health Organization. In the continental U.S.A., surface and barrier sprays are being considered using machinery adapted to produce a larger spray spectrum than space sprays. Some companies are marketing air-assisted sprayers for coverage of vegetation in barrier sprays. Although residual sprays are mentioned, the sheer diversity of equipment means that this should be addressed separately; detailed accounts can be found in documents highlighted under “Bibliography.”

The primary parameter in any application of sprays is droplet size, described by the droplet diameter measured in microns. The optimum droplet size for impact upon flying mosquitoes is in the range of 1–50 μm ; with 7–22 μm considered an optimum for both biological control and maximum dispersal in the air. For residual sprays, the droplets need to be small for good surface coverage, approximately 150 μm , but not too small as drift will become an issue.

THERMAL FOG APPLICATION

Thermal fogging employs LV technology (5–50 L/ha). During thermal fogging, the insecticide is diluted into an oil-based carrier liquid, and heat is used to decrease viscosity and vaporize the carrier and insecticide together. When the vapor is ejected and hits cooler air, it condenses to form a dense fog of droplets usually less than 3 μm , rarely more than 20 μm . The exact drop size is dictated by chemical formulation, flow rate, and temperature at the nozzle. Effective adulticiding using thermal fogging requires that the amount of time the insecticide is exposed to extremes of temperature is brief (fractions of a second), resulting in minimal degradation.

The advantage of thermal fogging is that it is easily visible, leading to good public relations, while the low concentration of the active agent in the mix reduces

operator exposure. The disadvantages are that large quantities of organic solvents are used which are expensive and have a disagreeable odor. In addition, there is a fire risk from machinery operating at very high temperatures with flammable solvents; thermal foggers must never be left unattended. Also low visibility in the fog can be a traffic hazard.

Vehicle-mounted thermal foggers are typically used to spray whole communities. This application technique requires a network grid of streets to effectively control mosquitoes. This is because, as with cold fogging, wind (<16 kph) is required to transport the insecticide. However, the effective downwind distance is rarely over 90 m. Thermal fog equipment uses an air-cooled motor to run a high-volume air blower, fuel pump, and insecticide pump. Air is delivered to the combustion chamber where it is mixed with gasoline vapor and ignited. Temperatures reach 426–648°C (800–1200°F) usually over 500°C. The diluted insecticide is pumped into a cup in the fog head, or directly into the nozzle, where it is then vaporized by hot gasses.

Hand-carried thermal foggers are used for treating houses and small outdoor areas. Two types are available—pulse jet and friction plate. Pulse jet applicators use batteries to initially ignite gasoline in a combustion chamber, with the hot exhaust gas igniting subsequent charges of fuel and air. A pulse jet engine will continue to operate as long as fuel is supplied through the carburetor. Insecticide is injected into the hot exhaust gas via a fixed restrictor, which controls the flow at rates up to 25 L/hr. A safety valve stops the flow of insecticide when the engine ceases to operate. Friction plate applicators use a 1–3 hp two-stroke engine, which drives a friction plate inside the insecticide tank. This preheats the insecticide and fuel oil mixture. The plate also serves as part of the pump that delivers the liquid to the engine exhaust where the hot exhaust gasses generate and disperse the fog. Friction devices operate at a lower temperature than pulse jet.

ULV (COLD FOGGING) APPLICATION

ULV technology, defined by the Environmental Protection Agency, is a method of dispensing insecticide in volumes less than 5 L/ha. The application of low volumes returns massive economic savings and significantly improves the logistics of pesticide applications.

Cold foggers mechanically break up the spray mixture, using high-speed rotary nozzles or high-pressure gaseous energy nozzles that produce an aerosol. The volume of spray is kept to a minimum, and concentrated ULV formulations are used. The advantages of ULV are that the diluent is kept to a minimum, and can be supplied ready to use, therefore keeping

operator exposure and handling costs to a minimum. Water-based or diluted formulations can be used, but oil-based is more common. Because heat is not involved, there is no risk of fire and no organic solvents are required and so this application technique is more environmentally friendly. The disadvantages are that dispersal of the spray cloud is difficult to observe and greater technical skills are required for more regular and precise calibration.

Nozzles capable of creating ULV aerosols are varied. Centrifugal energy nozzles produce droplets when the liquid is thrown from a rotating surface by centrifugal force. Increased rotational speed and a decrease in flow rates can create aerosol sprays. Examples of such nozzles are the spinning disc, cylinder, or rotary cage. These nozzles are not as common, however, as the gaseous energy nozzles where air velocity is used to shear the spray into aerosol droplets.

Small hand-carried cold fog machines typically have a 1–3 hp two-stroke gasoline engine (electric engines are also available), which drives a blower unit to discharge air through a nozzle. The air might also be used to slightly pressurize the insecticide formulation tank so that liquid is fed via a restrictor to the nozzle. Alternatively, negative pressure is generated by the airflow passing the nozzle, allowing liquid to be drawn from the tank. Most handheld cold foggers feed liquid into the airstream inside (centrally within) a tubular duct to feed to a vortical nozzle for more even atomization. Knapsack mist blowers weigh 11–25 kg and in most cases use a high velocity jet of air to create a “mist.” Some have a rotary nozzle mounted in the airstream. Flow rates are 1–4 L/hr, ideal for indoor or small areas outside.

Vehicle-mounted cold foggers use a 5–20 hp four-stroke gasoline engine (electric engines are also available) to drive a high-volume air blower, forcing air at a rate of approximately 6 m³/min at 50 kPa to one or more nozzles. Alternatively, a high-pressure low-volume air source is used with an air compressor rather than a blower. The pesticide container may be pressurized to force the formulation to the nozzle, or a positive displacement pump can be used. Positive displacement pumps are usually linked electrically to the vehicle to vary output as a function of vehicle speed. In particular, spraying ceases when the vehicle stops. The angle of projection can be adjusted for the task at hand. The nozzles used on the larger truck sprayers are generally vortical nozzles. Vortical nozzles utilize a low-pressure airstream from a compressor or blower so that the air is directed over a series of fixed vanes to produce a rotary movement or vortex, which increases the shearing action on a liquid. Average droplet sizes by volume of less than 25 µm are usually produced. Vortical nozzles have large orifices that seldom get blocked.

CONCLUSIONS

Ground ULV spraying and the choice of equipment are dictated by the task at hand. Today cold fogging is more popular than thermal fogging as it is economically, logistically, and environmentally more friendly. Handheld equipment are often reserved for small-scale application to a residence or a small community area. Truck spraying is typically used for larger scale applications in areas with extensive road networks.

Pesticide application is a multidisciplinary subject; effective adulticide applications are reliant on many aspects, including the equipment. One of the most important factors is meteorology; for example, wind (between 3 and 16 kph) is necessary to move chemicals to the desired location. In addition, an understanding of the biology of the target insect and the chemistry of the compound to be applied is required. Only then can pesticides be properly targeted and the proper application executed for effective control.

The following texts have been selected as valuable “Bibliography.”

BIBLIOGRAPHY

- American Mosquito Control Association. *Ground Equipment and Insecticides for Mosquito Control*; AMCA Bulletin No. 2, 1968.
- Matthews, G.A. *Pesticide Application Methods*, 3rd Ed.; Blackwell Scientific: Oxford, England, 2000.
- World Health Organization. *Equipment for Vector Control*, 3rd Ed.; Geneva, World Health Organization, 1990.
- World Health Organization. *Manual for Indoor Residual Spraying Application of Residual Sprays for Vector Control*; World Health Organization (WHO), Communicable Disease Control Prevention, and Eradication, WHO Pesticide Evaluation Scheme, 2002.
- World Health Organization. *Space Spray Application of Insecticides for Vector and Public Health Pest Control a Practitioner's Guide*, Pesticide Evaluation Scheme, World Health Organization, 2003.

Ethics, Biotechnology, and Pesticides

Hugh Lehman

University of Guelph, Guelph, Ontario, Canada

INTRODUCTION

In earlier works, I have attempted to explain some elements of ethical theory to show how ethical principles may be applied in efforts to resolve ethical controversy regarding pesticide use.^[1] Here I shall turn directly to the question of whether it is ethically acceptable to use genetic engineering to modify living organisms in order to control pests such as weeds, insects, or fungi. By the term “genetic engineering,” I refer to the use of techniques of molecular biology to deliberately insert genetic material taken from one organism, which we will call the *donor*, into another organism, which we shall call the *recipient*. Genetic engineering, as I understand the term, does not apply to use of other techniques that have been used to modify genomes or genotypes of organisms, such as selective breeding or mutagenesis. Conceivably, some of the considerations that we apply here to the use of genetic engineering in pest control would apply to other uses of genetic engineering also. Examples of the use of genetic engineering in the control of pests are the development of organisms that are genetically engineered to survive the application of herbicides and also the development of organisms that produce substances that are toxic to pests. Genetically engineered crops (genetically modified organisms, or GMOs) that are already in use include corn, soybeans, canola, and cotton. Genetically engineered wheat and other crops will be made available to farmers and consumers shortly.

Although the use of genetic engineering could produce many agricultural benefits, including crop varieties that will grow under growing conditions that are, at present, unfavorable for crops, we shall focus on the use of genetic engineering to control weeds, insects, or other agricultural pests. Genetically modified organisms for these latter purposes have already been approved and marketed even though serious questions have been raised about them concerning matters of food safety and environmental damage. These matters indicate a potential for serious harm to people or other creatures. Strong ethical arguments can be given for saying that people should not be exposed to these risks. These arguments support the contention that genetically engineered crops should

not be approved by our governments for use in agriculture until these crops have been shown, through rigorous scientific experimental procedures and public discussion of relevant ethical matters, to be safe enough for their intended uses. This conclusion can be grounded on utilitarian assumptions or, alternatively, on Kantian or Christian ethical assumptions. Given that there are safe alternatives to the current uses of GMOs, taking risks of serious harm without extensive and rigorous scientific experimentation is unlikely to produce a maximum satisfaction of desires or minimum frustration. Again, taking such risks without prior approval of the people who will be exposed to the risks is arguably incompatible with the Kantian principle that people ought to be treated with respect. Even the fundamental principle of Christian ethics, namely that we ought to love our neighbors, appears to imply that we ought not to subject them to risks of serious harm that could be avoided by taking greater care. A question has been raised as to whether governments have proceeded with appropriate caution in this matter. Let us pursue this question in somewhat greater detail.

PART 1

The creation of a GMO involves the insertion of a packet of genetic material into the deoxyribonucleic acid (DNA) of the organism. Depending on the intention of the producer of the GMO, the packet contains genetic instructions for a trait that one wants to be expressed in the organism (e.g., the trait might be a capacity to tolerate exposure to herbicides). The packet also contains other genetic materials that serve to control the activation of the DNA for the trait in question. Further, at the present time, the packet may contain antibiotic resistance genes, the products of which can be used to determine which plants have been genetically engineered. (In light of concerns about the spread of microorganisms that are resistant to antibiotics, efforts are currently underway to find methods of selection of genetically engineered plants that do not make use of genes for antibiotic resistance.)

The insertion of the packet of genetic material involves many unknowns. The person who inserts

the DNA packet is not able to control or determine precisely where in the genome of the recipient organism the packet will be situated. One is not able to anticipate precisely how the inserted DNA will interact with the other parts of the recipient's DNA. Thus there may be unanticipated gene products. Furthermore, processes that normally occur in unmodified organisms of the species or variety in question may be disrupted. Unanticipated gene products or other functional disruptions may not be immediately apparent, but may well result in undesirable consequences for the GMO itself or for some other organism that consumes the GMO. For example, the GMO may contain substances that are toxic to the persons or animals that consume them, or there may be reductions in quantities of nutrients.^[2] Furthermore, if there are plants growing in the neighborhood of the GMO that are sufficiently closely related to the GMO, then genetic material from the GMO can be spread to other organisms either through a reproductive interaction between the GMO and its close relatives, or through an interaction with microorganisms.^[3,4] For example, the tolerance of a herbicide that is engineered into a grain crop may be spread to other grasses that farmers regard as weeds. Indeed, this has already happened with some frequency. Where this happens, the herbicide that the organism has been engineered to tolerate loses its effectiveness.^[5] Crops genetically engineered to tolerate herbicides may spread widely from the fields in which they were planted and become weeds in other fields—weeds that cannot be controlled by the herbicide in question. In addition, in some cases where particular pests have been controlled through genetic engineering, other pests may proliferate. Thus genetic modification of organisms poses risks of serious harm to the health of consumers and to the well being of farmers and others.^a

Given the possibility of serious harm to consumers, serious damage to farmers through the creation of herbicide resistant weeds, and serious damage to many people through a disruption of relationships among organisms that constitute our common environment, it is incautious to introduce GMOs into farming or other ecosystems unless there are great benefits to be gained. To balance the risk of causing illness or injury, there would have to be potential gains of equivalent importance. Perhaps, to the balance risk of damage to ecosystems, there should be potential gains in avoiding further damage or in repairing systems that are already damaged. If harm is done through the development or use of GMOs, then those who are harmed

deserve compensation for the harm they suffered from those who caused the harm.

Supporters of the use of GMOs have alleged that the risks of harm from the use of GMOs are not significant because many of our other foods contain substances that pose risk of harm.^[7] Furthermore, supporters of the use of GMOs in agriculture have alleged that there are great benefits to be gained from the introduction of such products.^[8] They have argued that there is no significant risk of injury to consumers of food substances that contain products of genetically modified crops because foods containing such products have been shown to be *substantially equivalent* to the foods from which the GMOs are derived.^b They have argued that use of GMOs will lead to reductions of the amount of synthetic chemical pesticides in agriculture and, consequently, to reduction in harm associated with the use of such pesticides.^[11]

If GMOs really did not pose significant risks of harm, or if the probability that these benefits would be realized was high enough, or if the benefits were sufficiently great, then these claims would indeed constitute a strong reply to the criticism that GMOs have been marketed without due caution being taken. However, pointing out that other accepted foods pose significant risks of harm does not show that GMO foods pose no significant risks of harm. If other accepted foods pose significant risks of harm, then members of the public should be so informed and consideration should be given to discontinuing the acceptance of such other food. The fact that risks were taken through the introduction of crops produced by other techniques, such as selective breeding or mutagenesis, is not sufficient reason for taking significant risks by introducing GMOs. Conceivably, those earlier risks ought not to have been taken. Perhaps, as a society in prior times, we were not aware of taking such serious risks. Now that we have greater knowledge, we know how to be more careful and, as we have indicated above, there are strong ethical reasons for taking such care. Furthermore, GMOs such as herbicide-tolerant grains were introduced even though strong scientific evidence showing that there would be great reductions in the use of synthetic chemical pesticides was and is lacking. Agronomic data indicate that in many cases, the use of GMOs in agriculture is associated with no reduction, or even with increases, in the amount of synthetic chemicals applied.^[12] Insect pests can rapidly become resistant to crop plants that contain genetically engineered insecticides such as Bt. Even if the use of some genetically engineered foods were successful in

^aFor an extensive discussion of environmental risks, see Ref.^[6].

^bFor discussion and criticism of the use of the concept of substantial equivalence by supporters of the use of GMOs, see Ref.^[9]. Furthermore, see Ref.^[10]. For other references, see Ref.^[3].

bringing about reductions in the use of environmentally damaging pesticides, this benefit would not compensate for consumers taking the risk of consuming genetically modified foods that contain gene products in virtue of which a plant is herbicide-tolerant or toxic to insects. Furthermore, the claim that products that contain substances derived from GMOs will not harm consumers because they are substantially equivalent to foods that do not contain products of genetic modification is not a rigorously validated scientific claim. Either rigorous scientific experiments to establish safety have not yet been done or, if they have been done, the results have not been published as peer-reviewed papers in highly respected scientific journals.^[13] There can be impressive differences in composition among several genetically modified food products, each of which is substantially equivalent to a parental crop from which they are each derived. A GMO, which has been found to be substantially equivalent to the crop from which it was derived, might nonetheless contain substances that make it harmful to consume—substances that are not present in the parental crop. Furthermore, there can be significant differences in the values of various nutrients.

CONCLUSION

Supporters of the use of existing genetically modified crops in agriculture have not made satisfactory replies to the criticism that the introduction of such crops is ethically unacceptable because the manner of the introduction was incautious. Published scientific papers do not validate their claims about consumer or environmental safety. Thus, we conclude that crops such as those to which we referred above should not have been approved for use. Conceivably, such approvals should be withdrawn pending the exercise of due caution. The use of GMOs to assist farmers in controlling weeds, insects, or other pests could be ethically acceptable provided that due precautions are taken. Many other ethical questions need to be addressed concerning the use of GMOs in agriculture. We have restricted our discussion primarily to considerations relating to herbicide-tolerant GMOs and to GMOs that produce insecticides.

REFERENCES

1. Lehman, H. Ethical aspects of pesticide use. In *Encyclopedia of Pest Management*; Pimentel, D., Ed.; Marcel Dekker, Inc.: New York, 2002.
2. Ewen, S.; Pusztai, A. Effect of diets containing genetically modified potatoes expressing *Galanthus nivalis* lectin on rat small intestine. *Lancet* **1999**, *354* (9187).
3. Hopkin, K. The risks on the table. *Sci. Am.* April **2001**, *284* (4), 60–61.
4. Mellon, M. Does the world need GM foods? *Sci. Am.* April **2001**, *284* (4), 64–65.
5. Hall, L.; Topinka, K.; Huffman, J.; Davis, L.; Good, A. Pollen flow between herbicide-resistant *Brassica napus* is the cause of multiple-resistant *B. napus* volunteers. *Weed Sci.* **2000**, *48*, 688–694.
6. Letourneau, D.K., Burrows, B.E., Eds.; *Genetically Engineered Organisms: Assessing Environmental and Human Health Effects*; CRC Press: New York, 2002.
7. Prakash, C.S. The genetically modified crop debate in the context of agricultural evolution. *Plant Physiol.* **2001**, *126*, 8–15.
8. Bond, C. The benefits and politics of biotechnology. In *Speech to U.S. Senate*; January 26, 2000.
9. Clark, E.A.; Lehman, H. Assessment of GM crops in commercial agriculture. *J. Agric. Environ. Ethics* **2001**, *14* (1), 3–28.
10. February 5, 2001. Available at: www.rsc.ca.
11. Brown, K. Seeds of concern: are genetically modified crops an environmental dream come true or a disaster in the making? Scientists are looking for answers. *Sci. Am.* April **2001**, *284* (4), 52–57. This article also gives further references.
12. Ryan, A. *To Bt or Not to Bt*; [//www.biotech-info.net/to_Bt_or_not_to_Bt.html](http://www.biotech-info.net/to_Bt_or_not_to_Bt.html).
13. Domingo, J.L. Health risks of GM foods: Many opinions but few data. *Science* **2000**, *288*, 1748–1749.

BIBLIOGRAPHY

- Lehman, H. Environmental ethics and pesticide use. In *Techniques for Reducing Pesticide Use: Economic & Environmental Benefits*; Pimentel, D., Ed.; John Wiley & Sons: Chichester, England, 1997; 35–50.
- Lehman, H. *Rationality and Ethics in Agriculture*; University of Idaho Press: Moscow, ID, 1995.
- Lehman, H. Values, ethics and the use of synthetic pesticides in agriculture. In *The Pesticide Question: Environment, Economics and Ethics*; Pimentel, D., Lehman, H., Eds.; Chapman and Hall: New York, 1993; 347–379.

Evolved Herbicide Resistance: Fitness Costs

Martin Vila-Aiub

Western Australian Herbicide Resistance Initiative (WAHRI), School of Plant Biology, Faculty of Natural and Agricultural Sciences, University of Western Australia (UWA), Crawley, Western Australia, Australia

Paul Neve

Western Australian Herbicide Resistance Initiative (WAHRI), University of Western Australia (UWA), Crawley, Western Australia, Australia

Stephen Powles

Department of Agriculture, Western Australia Herbicide Resistance Initiative (WAHRI), University of Western Australia (UWA), Crawley, Western Australia, Australia

INTRODUCTION

Herbicides have been intensively employed for the last 50 years to manage weed infestations in agricultural fields.^[1] As a result of frequent use and the high selection pressure that herbicides impose, herbicide-resistant weed populations have evolved worldwide. Alleles conferring herbicide resistance arise in weed populations by random spontaneous mutation and are present at very low frequencies before herbicide selection. However, given the extraordinary advantage they confer, these alleles are rapidly selected in weed populations under herbicide selection.^[1]

It has been often assumed that organisms with heritable resistance to environmental stresses will exhibit an ecological disadvantage (i.e., termed fitness cost or cost of resistance) compared to susceptible organisms when the selective force or stress is absent.^[2] This theory predicts that herbicide resistance comes at an ecological and/or physiological cost.

BASIS FOR FITNESS COSTS ASSOCIATED WITH HERBICIDE RESISTANCE

The resource-based allocation theory predicts that plants divert resources to different organs and functions in order to maximize their adaptive strategy according to the selection imposed by the environment.^[3] Given the fact that resources are limited, any increase in allocation to one organ or function implies a decrease in allocation to other sinks. This theory is exemplified by the trade-off usually found in plants between reproduction, growth, and storage or defense functions. Herbicide sequestration and detoxification are herbicide resistance mechanisms (non-target-site resistance mechanisms) found in many weed species.^[4] These mechanisms are mediated by proteins (enzymes

and membrane transporters) that rapidly metabolize and/or sequester a herbicide away from its site of action. According to the resource-based allocation model, further energy investments will be required to synthesize these proteins, resulting in diversion of these resources away from other functions and therefore a fitness cost in the absence of the herbicide.

A second concept supporting the fitness cost hypothesis is that the selection and fixation of adaptive mutant alleles in plant populations may have detrimental pleiotropic effects on normal plant function or metabolism. In this case also, the mutant form may suffer a relative physiological/ecological disadvantage compared to the wild type in the absence of selection.^[5] The majority of cases of herbicide resistance result from nucleotide mutations in genes encoding enzymes that are herbicide targets (target-site resistance mechanisms). The amino acid substitutions that result from nucleotide mutation usually change the dimensional configuration of the enzyme, preventing or reducing effective herbicide binding. Such mutations may therefore result in a fitness cost by compromising normal enzyme function by interfering with the regulation of metabolic pathways, or by other unknown pleiotropic effects.

IMPORTANCE OF FITNESS COSTS ESTIMATION

The assessment of fitness costs in herbicide-resistant weed populations has ecological and agronomic implications. Clearly, the presence and extent of any fitness cost will influence the population dynamics of resistance evolution and regression after herbicide selection pressure is removed. Estimates of differential fitness between resistant and susceptible genotypes in the absence of herbicides are key inputs for simulation models that predict the population dynamics and

genetics of herbicide resistance evolution. These models can be used to design and test management strategies for the minimization and management of herbicide resistance. The likelihood and rapidity with which a resistant population may revert to susceptibility (resistance regression) when herbicide use ceases or is temporarily suspended is influenced by the existence and magnitude of fitness costs.^[6] Where there is a fitness cost, this rate of regression will determine the success of weed control programs which attempt to combat resistance by exploiting fitness costs (e.g., different crops/pasture rotations, fallow, alternative herbicides). In this way, the management of herbicide resistance benefits from fundamental knowledge of the physiology, ecology, and genetics of herbicide resistance. Understanding the differential life history traits of susceptible and resistant types can be used to design and implement management practices which exploit these differences to favor the susceptible type, thus delaying or reversing the spread of resistance.^[6]

PHYSIOLOGICAL FITNESS COSTS RELATED TO TARGET-SITE RESISTANCE

The best-documented case is one in which a target-site mutation endows resistance but a substantial fitness cost is resistance to the triazine herbicides due to the *psbA* mutation. Triazine herbicides are toxic to plants because they inhibit photosynthesis. In photosystem II (PSII), carbon fixation is driven by a series of oxidation and reduction reactions that promote electron transport between proteins located in the chloroplasts. As part of this process, in PSII, electrons are transferred from plastoquinones Q_A to Q_B . After electron transfer and acceptance of protons, fully reduced plastoquinone Q_B leaves the Q_B site on the D1 protein to donate electrons to the cytochrome b_6/f complex. Triazine herbicides are potent PSII inhibitors with a high affinity for the D1 protein and competitively inhibit plastoquinone Q_B binding, preventing electron transport and therefore inhibiting the photosynthetic process. In many parts of the world, resistance to PSII-inhibiting herbicides has evolved as a result of a point mutation in the chloroplastic *psbA* gene encoding D1 protein. This single-nucleotide change results in the substitution of serine 264 for glycine in the D1 protein and reduces the affinity of PSII herbicides at the Q_B site. However, this mutation reduces the rate of electron transfer between Q_A and Q_B plastoquinones, thereby reducing photosynthetic efficiency.^[7] These plants benefit greatly from the mutation in the presence of triazine herbicides but pay a fitness cost in the absence of these herbicides. This reduced photosynthetic potential has dramatic effects at the individual plant and population levels. Low growth rates,

compromised competitive ability, and reduced seed production have commonly been observed in *psbA* gene-endowed triazine-resistant compared to triazine-susceptible weed biotypes.^[8] Similarly, canola (*Brassica napus*) crops with resistance to triazines due to the *psbA* gene regularly display a 10–40% yield reduction compared to non *psbA* gene cultivars. Simulation models and field observations have shown that resistance costs associated with resistance to PSII inhibitors can lead to a decline in the frequency of resistant plants over a few generations when PSII-inhibiting herbicides are not used.

As described above it is clear that triazine resistance endowed by a specific mutation of the *psbA* gene results in an ecological fitness cost to the plant. However, it should not be concluded that other cases of herbicide resistance will always involve a fitness penalty. Indeed, each case needs to be individually investigated. Many weedy species have evolved target-site-based resistance to the acetolactate synthase (ALS)-inhibiting herbicides and several different resistance-endowing mutations have been identified.^[9] A number of studies which have evaluated fitness of target-site-based plants resistant to ALS-inhibiting herbicides do not report a fitness penalty associated with ALS herbicide resistance.^[9] However, for the proline (197) to serine mutation of the ALS gene, Bergelson has reported a fitness penalty.^[10] As there are many mutations of the ALS gene that can endow herbicide resistance, it is entirely possible that some of these mutations result in no or very small fitness penalties, whereas other mutations endow substantial fitness penalty. The same is true for the acetyl CoA carboxylase (ACCase)-inhibiting herbicides in which several studies do not report a fitness penalty.^[8] However, as several mutations of the ACCase gene which endow herbicide resistance are now known, fitness studies will need to be conducted for each of these mutations. Similarly, non-target-site-based mechanisms can endow resistance to the triazine, ALS, and ACCase herbicides and in these cases no fitness studies have been conducted to find out whether or not the non-target-site mechanisms (enhanced metabolism) endow any fitness penalty.

It is emphasized that it is not possible to generalize as to whether or not a particular resistance mechanism will or will not result in a fitness cost. Whether or not a particular mutation which endows herbicide resistance will also express a resistance cost awaits definitive studies for each mutation. Unfortunately, it is often not possible to conduct such fitness studies because resistance mechanisms and their genetic control are sometimes unknown or require much research before they are known. Therefore, researchers conduct as best they can a fitness study without knowledge as to the resistance mechanism or its genetic control.

CONCLUSIONS

Theoretical support for the existence of fitness costs associated with herbicide resistance in weeds is based on resource allocation theory and considerations of the pleiotropic effects of herbicide target-site mutations. These fitness costs, where they exist, can assist in the management of herbicide-resistant weeds. This contribution has reviewed the state of knowledge of the best-documented example of fitness costs associated with target-site resistance (triazine resistance). Further research efforts are needed to unequivocally assess the existence of fitness penalties in other herbicide resistance cases. To date, there is no empirical evidence to reject or accept the resource allocation theory, which predicts resistance costs in weeds with non-target-site resistance. This highlights an important gap in current knowledge. Future studies should also seek to better understand the ecology of herbicide-resistant weeds as this knowledge can benefit the design and implementation of management to prevent, delay, or regress herbicide resistance.

REFERENCES

1. Powles, S.B.; Preston, C.; Bryan, I.B.; Jutsum, A.R. Herbicide resistance: impact and management. *Adv. Agron.* **1997**, *58*, 57–93.
2. Fisher, R.A. *The Genetical Theory of Natural Selection*; Dover: New York, 1958.
3. Harper, J.L. *Population Biology of Plants*; Academic Press: London, 1977.
4. Powles, S.B.; Holtum, J.A.M. *Herbicide Resistance in Plants. Biology and Biochemistry*; Lewis Publishers: Boca Raton, FL, 2001.
5. Uyenoyama, M.K. Pleiotropy and the evolution of genetic systems conferring resistance to pesticides. In *Pesticide Resistance: Strategies and Tactics for Management*; National Academy Press: Washington, DC, 1986; 207–221.
6. Diggle, A.J.; Neve, P. The population dynamics and genetics of herbicide resistance. A modeling approach. In *Herbicide Resistance and World Grains*; Powles, S.B., Shaner, D.L., Eds.; CRC Press: Boca Raton, FL, 2001; 61–99.
7. Gronwald, J.W. Resistance to photosystem II inhibiting herbicides. In *Herbicide Resistance in Weeds. Biology and Biochemistry*; Powles, S.B., Holtum, J.A.M., Eds.; Lewis Publishers: Boca Raton, FL, 1994; 27–60.
8. Holt, J.S.; Thill, D.C. Growth and productivity of resistant plants. In *Herbicide Resistance in Weeds. Biology and Biochemistry*; Powles, S.B., Holtum, J.A.M., Eds.; Lewis Publishers: Boca Raton, FL, 1994; 299–316.
9. Tranel, P.J.; Wright, T.R. Resistance of weeds to ALS-inhibiting herbicides: what have we learned? *Weed Sci.* **2002**, *50*, 700–712.
10. Bergelson, J.; Purrington, C.B.; Palm, C.J.; Lopez-Gutierrez, J.C. Costs of resistance: a test using transgenic *Arabidopsis thaliana*. *Proc. R. Soc. Lond. B* **1996**, *263*, 1659–1663.

Facultative Predation as a Biological Control

Oscar Alomar

Department of Protecció Vegetal, IRTA, Cabrils (Barcelona), Spain

INTRODUCTION

Predators that can facultatively feed on plants are distributed among several arthropod taxa. Some have been considered pests and thus targeted in control programs. However, increasing interest in conservation and augmentation of native fauna in biological control has led to regarding such facultative predators as potentially useful if management programs are able to integrate their benefits and risks.

OMNIVORY, ZOOPHYTOPHAGY, AND PHYTOZOOPHAGY

Most arthropods are considered to belong to one of the trophic categories; zoophagous (consuming animal materials) or phytophagous (consuming plant materials). However, many are instead omnivores, consuming both.^[1-4] In fact, there is a continuum of feeding strategies. Phytophagous insects are known to consume other insects (e.g., the western flower thrips is also a mite predator). Conversely, several predatory arthropods also feed on plant products (e.g., nectar, pollen, plant juices, developing seeds) at some stage during their life cycle. The extent to which feeding at another trophic level is occasional, opportunistic (e.g., to compensate for nutritional deficiencies in their normal prey), or necessary is not known in most cases.^[5] In most arthropod species, there is also a switch between predatory and plant feeding habits at different life stages (e.g., hoverflies and lacewings with predaceous larvae and plant-feeding adults). But both trophic levels can also be consumed during the same life stage. The term facultative is used to indicate consumption of materials at another trophic level within the same developmental stage. The terms zoophytophagy and phytozoophagy are also used, depending on one's perception of their relative position in this continuum of feeding habits.

Benefits of Plant Feeding by Predators

The significance of plant feeding has often been overlooked by biological control specialists, and many biocontrol attempts have failed as a result. Traditionally, the consumption of plant materials by predators has been regarded more as peripheral to what is considered the most important aspect: predation. However, many

predators require foods that are often not available in crops, and consumption of plant foods may provide nutrients that are either essential for their diet or are a substitute resource when prey are scarce, and therefore play a critical role in maintaining predators.

Facultative feeding habits are present in several predaceous groups.^[1,6] Coccinellids often turn to pollen or nectar as food when prey becomes scarce. Many Phytoseiid mites can readily be maintained and produce viable offspring by feeding on pollen, and plant exudates can serve as food supplements. Establishment of minute pirate bugs (*Orius* spp.) is not possible on cucurbits that lack pollen. Feeding on both plant and animal materials by Carabids is probably more significant than is acknowledged. Within the Heteroptera, the use of a broad range of both animal and plant food sources appears to be widespread:^[7-9] supplementing prey diet with plant material accelerates nymphal development, increases nymphal and adult longevity and survival, and enhances fecundity. The benefits derived from such feeding habits are species-specific and depend on the quality of prey and plant components in the diet. That facultative plant feeding is a successful adaptation is shown by the ubiquity in many crops of Heteroptera using such a feeding strategy: They can be found in orchards, alfalfa, cotton, and especially in annual crop systems (e.g., soybean and tomato), where this adaptation permits subsistence under unpredictable fluctuations of prey abundance.

Feeding on plants should not then be seen to limit the potential effectiveness if compared with more voracious predators, but rather to provide necessary resources to sustain themselves on fewer prey.^[7-8,10] Such predators will be best suited to establish and subsist at low and infrequent prey meals, which are necessary for successful biological control; i.e., get established early in the crop cycle in order to prevent or retard build-up of high numbers of residents, and also of new exotic pests. Their contribution to biological control is prevention—providing steady levels of mortality—rather than eradication of pest outbreaks.^[11]

Risks of Plant Feeding—Incorporation into IPM recommendations

Feeding on plant liquids can be detrimental to the predator if the crop is treated with systemics, which

have minimal impact on other predators. Early researchers also soon realized the ambivalent role of some facultatively phytophagous predatory Heteroptera, and that their beneficial value may be counteracted by the economic damage some may cause as a herbivore (e.g., feeding on fruits or plant tissue, by their ovipositorial activity or risk of transmitting plant pathogens).

Therefore, an understanding of how and when damage is likely to occur is necessary before management decisions can be made, but our biological control strategy also has to be considered.

Conservation biocontrol

Most of the work with facultative predators has been related to conservation biocontrol programs in orchards, cotton, and vegetables.^[7,8,11–13] In this situation, facultative predators are members of a complex of other predators that spontaneously enter the crops. *Campylomma verbasci* (Meyer-Dür) and *Atractotomus mali* (Meyer-Dür) are important components of the overall complex of aphid predators on pome fruits, and also prey on psylla and spider mites. Both can injure the fruit of some apple cultivars, but then only during key phenological windows (after bloom), and not in all regions. In other periods, and with other apple cultivars and most pears, they are not considered a pest and are exploited for their predaceous benefits. Moreover, the degree of fruit damage is not simply related to population density of the predator, but is also affected by the availability of other foods: pollen, nectar, plant juices, or animal prey.

The development of IPM for tomato crops in the Mediterranean basin also exemplifies the usefulness and management of facultatively phytophagous predators.^[11] *Dicyphus* species are efficient predators in vegetable crops, but may also blemish tomato fruits. Damage by *Dicyphus tamaninii* Wagner in commercial fields has been related to high predator-to-prey ratios: injury increases once whitefly is brought under control, and less damage is recorded if enough prey are available. A decision chart advises when to spray, and has resulted in a substantial reduction in insecticide use without resulting in *Dicyphus* or whitefly damage. *Nesidiocoris tenuis* (Reuter) also greatly reduces whitefly populations, but, again, damages flowers and young shoots once prey is depleted and poses a problem for long-season tomato greenhouses when pest levels are reduced by other entomophagues.

Augmentation biocontrol

Augmentation biocontrol presents a different setting to discuss facultative plant feeding by predators. Here, the decision is whether or not to enhance the numbers

of a known useful entomophage when spontaneous occurrence originating from outdoor is too late to be of practical importance, or nonexistent. The management of *Macrolophus caliginosus* (Wagner) is a valid model.^[11] Spontaneous colonization of field and greenhouse tomatoes in the Mediterranean basin has not resulted in injuries, and *Macrolophus* has been commercially available and widely released in European tomato greenhouses for several years. However, injury has been claimed in northern Europe on cherry tomatoes, but always at the end of the crop, with high numbers of the predator, and when hardly any prey are left. This shows, again, that management of facultative predators is far from simple, and that we cannot just blame the predator or question its status. The benefits of a predator such as *M. caliginosus* are not in question. We simply need a better understanding of all aspects of their use, and to determine the situations when damage is likely. Current work with *D. hesperus* (Knight) in British Columbia, Canada addresses its contribution to whitefly control on tomato vs. blemishing of fruits.^[14]

Damage relationships are not simple and, as with all pests, depend on species, stage, and abundance of both predator and prey, and on crop and cultivar (e.g., light- vs. dark-skinned apples). Biocontrol practices must avoid high predator-to-prey ratios at susceptible growth stages on susceptible cultivars. Most facultative predators perform better on mixed diets (e.g., when there is an abundance of insect prey). Therefore, late releases when pest populations are already established will lead to large numbers of the predator when prey is controlled and to a risk of damage, especially in enclosed environments. Commercial requirements for almost “sterile” crops should also not seek inundative releases of such predators even if pests are present, as this may force them to feed on growing plant parts as fruits. Although these predators are generalists, they should not be applied as a cure-all treatment against every pest in the crop: low-quality prey may increase plant feeding to compensate for poor nutrients. Use in other crops should also be tested before release. *D. tamaninii* does not cause fruit injury in hard-skinned cucumber varieties,^[15] but does so on Dutch type cv., as happens with thrips scarring. Finally, mature crops, with older leaves of poor quality, may also be more injured as the predator shifts to feeding on growing parts.

Overall, detailed economic studies on the value of facultative predators are lacking. In conservation biological control, their value within the complex of predators is generally acknowledged for many crops, but no specific study has yet addressed the cost and benefit of each predator. Such dilemmas should be studied on a case-by-case basis.^[13,14,16] However, case studies as the colonization of field tomatoes by

facultative predators (e.g., *D. tamaninii*, albeit blemishing tomatoes), led to a reduction in the number of pesticide applications and greatly contributed to the implementation of IPM programs in the area.^[11] The sale of *M. caliginosus* for greenhouse crops and current work on other facultative predator species indicate the interest of industry in entomophagous that are also generalists and that may be used for the control of current pests and as a response to new exotic pest problems.

CONCLUSION

The key to the use of facultative predators in biological control lies in measuring their contribution to overall pest control, and determining the potential risks of plant feeding and the circumstances of diet shifts. To the extent that risk is demonstrated, specific management criteria have to be developed that simultaneously avoid injury, but still profit from their predation. Unnecessary decisions that interfere with the action of facultative predators may disrupt current levels of biological control and lead to pest resurgence.

REFERENCES

- Hagen, K.S.; Mills, N.J.; Gordh, G.; McMurtry, J.A. Terrestrial arthropod predators of insect and mite pests. In *Handbook of Biological Control*; Bellows, T.S., Fisher, T.W., Eds.; Academic Press: San Diego, 1999; 383–503.
- Whitman, D.W.; Blum, M.S.; Slansky, F. Carnivory in phytophagous insects. In *Functional Dynamics of Phytophagous Insects*; Ananthakrishnan, T.N., Ed.; Oxford and IBH Publishing: New Delhi, 1994; 161–205.
- Jervis, M.A.; Kidd, N.A.C. Phytophagy. In *Insect Natural Enemies*; Jervis, M., Kidd, N., Eds.; Chapman & Hall: London, 1996; 375–394.
- Obrycki, J.J.; Tauber, M.J.; Tauber, C.A.; Ruberson, J.R. Prey Specialization in insect predators. In *Radcliffe's IPM World Textbook*; Radcliffe, E.B., Hutchison, W.D., Eds.; University of Minnesota: St. Paul, Minnesota, 1997; <http://ipmworld.umn.edu/chapters/obrycki.htm> (accessed March 2002).
- Overmeer, W.P.J. Alternative Prey and Other Food Resources. In *Spider Mites. Their Biology, Natural Enemies and Control. A*; Helle, W., Sabelis, M.W., Eds., World Crop Pests; Elsevier: Amsterdam, 1985; Vol. 1B, 131–139.
- McMurtry, J.A.; Croft, B.A. Life-styles of phytoseid mites and their roles in biological control. *Annu. Rev. Entomol.* **1997**, *42*, 291–321.
- Alomar, O.; Wiedenmann, R.N., Eds.; *Zoophytophagous Heteroptera: Implications for Life History and Integrated Pest Management*, Proceedings Thomas Say Publications in Entomology: Lanham, MD, 1996; 1–202.
- Coll, M.; Ruberson, J.R., Eds.; *Predatory Heteroptera: Their ecology and use in biological control*, Proceedings Thomas Say Publications in Entomology: Lanham, MD, 1998; 1–233.
- Wheeler, A.G., Jr. Predacious plant bugs (Miridae). In *Heteroptera of Economic Importance*; Schaefer, C.W., Panizzi, A.R., Eds.; CRC Press: Boca Raton, 2000; 657–693.
- Lucas, E.; Alomar, O. *Macrolophus caliginosus* (Wagner) as an intraguild prey for the zoophytophagous *Dicyphus tamaninii* Wagner (Heteroptera: Miridae). *Biol. Control* **2001**, *20*, 147–152.
- Albajes, R.; Alomar, O. Current and potential use of polyphagous predators. In *Integrated Pest and Disease Management in Greenhouse Crops*; Albajes, R., Lodovica Gullino, M., van Lenteren, J.C., Elad, Y., Eds.; Kluwer Academic Publishers: Dordrecht, 1999; 265–275.
- Hull, L.A.; Horsburgh, R.L. Mite predators. In *Mid-Atlantic Orchard Monitoring Guide*; Hogmire, H.W., Jr., Ed.; Northeast Regional Agric. Engineer. Serv. Publ., 152 Riley-Robb Hall: Ithaca, New York, 1995; Vol. 75, 69–75.
- Reding, M.E.; Beers, E.H.; Brunner, J.F.; Dunley, J.E. Influence of timing and prey availability on fruit damage to apple by *Campylomma verbasci* (Hemiptera: Miridae). *J. Econ. Entomol.* **2001**, *94* (1), 33–38.
- Gillespie, D.; McGregor, R.; Quiring, D.; Foisy, M. Biological Control of Greenhouse Whitefly with *Dicyphus hesperus*. In *Agriculture and Agri-Food Canada, Pacific Agri-Food Research Station, Agassiz Technical Report, 2000*; No. 157 (PDF file at http://res2.agr.ca/parc-crapac/english/3electronic_publications/e_pubs.htm) (accessed March 2002).
- Castañe, C.; Alomar, O.; Riudavets, J. Management of western flower thrips on cucumber with *Dicyphus tamaninii* (Heteroptera: Miridae). *Biol. Control* **1996**, *7*, 114–120.
- Wilson, L.J.; Bauer, L.R.; Lally, D.A. Effect of early season insecticide use on predators and outbreaks of spider mites (Acari: Tetranychidae) in cotton. *Bull. Entomol. Res.* **1988**, *88*, 477–488.

Field Release, Captive Rearing for

Leonard Nunney

Department of Biology, University of California-Riverside, Riverside, California, U.S.A.

INTRODUCTION

Effective captive rearing of insects for field release is a deceptively complex task. The goal is generally clear: To facilitate the release of large numbers of high-quality individuals. However, achieving this goal is complicated by population genetic processes that can profoundly influence the value of the insectary colony as measured by the success of its field releases. This value depends sequentially on three major factors, the genetic composition of the founding population, the number of individuals reared (colony productivity), and the quality of reared individuals. The founding population defines the raw material on which all else depends, and the rearing techniques influence the quality of individuals. In general, optimizing the first step, the founding of the captive population, can be guided by the general principle that bigger is better. In contrast, optimizing the rearing strategy is complicated by the “paradox of captive rearing:” Adaptation to captivity generally increases productivity but at a cost of lower quality.^[1] This trade-off between quantity in the rearing facility and quality in the field should drive the management of captive populations prior to field release toward either optimizing the trade-off or minimizing it. Over 30 years ago, Boller^[2] expressed concern over the economic focus on quantity (cost per individual produced) rather than the true economic metric of effectiveness (the cost of achieving the intended goal). That concern is still relevant today.

THE FOUNDING POPULATION

To establish a successful breeding colony, the size of the initial sample is crucial. A bigger sample captures more of the natural genetic variability of the species. It also minimizes the risk of a deleterious “founder effect.”

The negative consequences of a founder effect stem from sampling error. Given a small founding population of size N , any disadvantageous allele carried by a founding individual is represented at an abnormally high frequency (as the minimum frequency is $\geq 1/2N$). Conversely, some of the beneficial variation present in the natural population at moderate frequencies will inevitably be lost.

Populations founded from a few tens of individuals are extremely prone to these pathological effects.

Beyond this size, a “large enough” founding number becomes more difficult to define, but a population size of $N > 1000$ is a good target for founding and maintaining a population for short-term captive rearing.^[3] Even when practical constraints preclude a single large initial sample, several samples of $N > 100$ of unrelated individuals are necessary to adequately capture the genetic variation of the original population.^[4]

The founding population should be sampled from the release site or from a region that is ecologically and climatically similar. However, there is no simple answer to the merits of the mixing population from different geographical locations to increase genetic variability. In some cases, mixing has proved successful for field release, but it can lead to the breakdown of geographically coadapted gene complexes leading to unpredictable phenotypic shifts in traits such as mating behavior, or a general loss of fitness.^[5] The best indicator of a potential problem is a large genetic distance among the sample populations. As such differences can occur over short geographical distances,^[5] the evaluation of genetic distance should routinely precede the mixing of different populations.

Having initiated a captive population with high genetic variability, there is a real danger of this variability being lost over the first few generations of captivity owing to a “crash-recovery” cycle.^[6] This cycle arises when a few genotypes are by chance preadapted to the rearing conditions, but the remainder reproduces very poorly. The result is a numerical bottleneck (the crash), driving a substantial loss of genetic variability. The problem can be reduced by subdividing the founders into very small breeding units; so the reproductive success of a broad range of genotypes is guaranteed. This strategy of subdivision may be adopted for just a few generations; however, there are often significant advantages to keeping the captive population highly subdivided until just prior to field release (see the following section).

THE PARADOX OF CAPTIVE BREEDING

Once a genetically variable captive population is successfully established, it might appear to be a relatively straightforward task to maintain a vigorous colony. Unfortunately, this is not the case. Genetically

variable captive populations rapidly adapt to their new environment—the rearing facility. This process is generally referred to as “domestication.” Domestication affects the gene pool of a captive population by actively favoring particular traits such as rapid development time;^[7] however, it can also have an effect by relaxing selection on traits under strong selection in the field. Relaxed selection typically increases trait variance, a pattern documented for predator-avoidance behavior in captive-bred mice.^[8]

Domestication drives the “paradox of captive breeding:” In general, it is not possible to maintain field quality while simultaneously increasing rearing quantity.^[1] For example, the mating success of male melon flies in the field showed a steady decline beyond 10 generations of captive rearing.^[1] This deterioration arises from a classic evolutionary trade-off whereby fitness in one environment is negatively correlated with fitness in a second. In this case, the two environments, field vs. rearing, differ across many important variables.^[4]

Given this trade-off, how should captive rearing be managed? There are two alternative strategies, optimization and avoidance.

The Optimization Strategy

The goal of the optimization strategy is to maximize the effectiveness (E) of a rearing facility. Effectiveness is the product of quantity (P = productivity of the rearing facility) and quality (w = per capita field success of released individuals), given that adaptation to the rearing environment (increasing P) will change (and generally decrease) w

$$E = Pw(P) \quad (1)$$

The optimum solution that maximizing E requires is:

$$\frac{dw(P)}{dP} = -\frac{w}{P} \quad (2)$$

which usually defines a partially domesticated population that provides the best balance between rearing productivity and field quality (Fig. 1). It is possible for the drop in field quality with domestication to be so rapid that no intermediate optimum exists,^[1] an outcome necessitating either a redesign of the rearing conditions, or the use of the avoidance strategy (see the following section).

Populations generally approach the relatively stable state of a “domesticated stock” (Fig. 1) within about 10–20 generations. Beyond this point, further evolutionary change should be relatively slow, driven

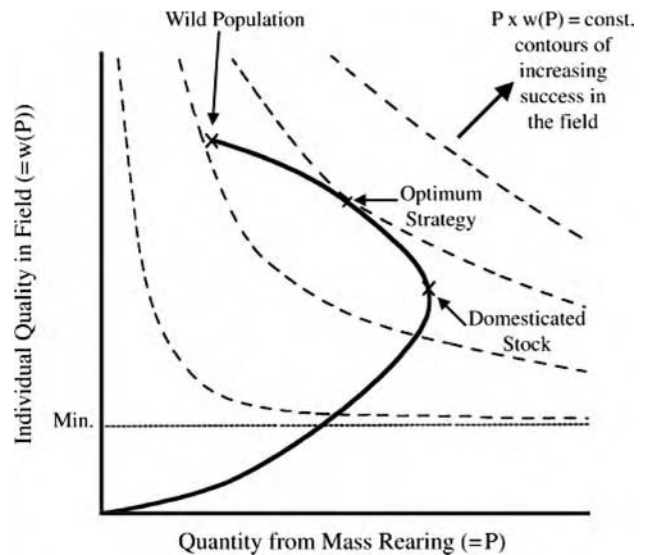


Fig. 1 The trade-off between numbers produced in captive rearing (quantity) and field performance (quality) during domestication. The solid curve defines the expected evolutionary path of the trade-off following capture and each dashed curve defines the combinations of quality and quantity that could achieve a given goal.

primarily by a gradual fixation of deleterious alleles owing to continuous inbreeding. This inbreeding reduces both quantity and quality in very old stock populations (the lower part of the solid trade-off curve in Fig. 1).

Given a long-term program of field release, one efficient approach to optimizing effectiveness is to sequentially establish new populations, with old populations discarded once they have evolved beyond the optimum (Fig. 1). For melon flies, this corresponds to an interval of about nine generations. It may be possible to increase the useful life of captive populations by incorporating field-related stimuli into the rearing facility, rearing stocks intermittently under seminatural conditions using field cages, or by recapturing released individuals and reintroducing them into the stock population.

A more direct way of keeping the captive stock close to the optimum is by continuously introducing wild-caught individuals. Introductions must be monitored to confirm that the relatively maladapted wild individuals are reproductively successful in the captive environment. One approach is to pair mate wild and captive individuals.

The Avoidance Strategy

An effective strategy for preventing adaptation to rearing conditions is to maintain the population as a

Table 1 Establishing and maintaining a captive population for successful field release

| Stage | Goal | Solution |
|--|--|---|
| Founding sample | Genetically variable population by size | Bigger is better. Sample $N > 1000$ or several samples of $N > 100$ |
| | Genetically variable population by mixing (optional) | Danger of outbreeding depression. Sample from several populations separated by a small genetic distance |
| | Population adapted to field release site | Sample on or near the field site. If not possible, sample from climatically and ecologically similar area |
| Establishment in captivity | Maintain genetic variation | Avoid “crash-recovery” bottleneck. Initially establish many subpopulations |
| Captive rearing: the optimization strategy | Maximize “effectiveness” [Eq. (1)] of future field release | Options: <ol style="list-style-type: none"> 1. Replace populations about every 10 generations 2. Continuously incorporates new wild-caught individuals 3. Incorporate rearing strategies that maintain field-related fitness |
| Captive rearing: the avoidance strategy | Avoid “domestication” during captive rearing | Isofemale lines—ideally maintain $\gg 100$ separate lines Lines must be intercrossed prior to field release |

large number of partially inbred (isofemale) lines.^[9] Inbred lines cannot adapt, and the avoidance strategy is the best approach when there is no intermediate optimum effectiveness, because quality is rapidly lost upon domestication.^[1] It is also the best approach when it is relatively easy to maintain multiple isofemale lines. An ideal goal is to maintain a large number ($\gg 100$) of lines, although this may not always be practical. A high number insures a high probability of retaining moderately rare alleles and to provide a buffer against the loss of some difficult to maintain lines.

The isofemale lines do not need to be rigorously inbred after their initial founding, which helps to avoid extreme inbreeding depression. Typically, inbreeding depression is less problematic in haplodiploid animals than in diploids.

Isofemale lines are unsuitable for release: They may exhibit low fitness, and the number of distinct genotypes is limited to the number of lines. Prior to release, the inbred lines should be systematically crossed to create a population of F1 hybrids. These F1 hybrids (or better still their F2 or F3 offspring, creating recombinant genotypes) can then be released.

CONCLUSIONS

The success of a field release using captive-reared individuals can depend crucially on the way in which the

captive population was established and maintained. Crucial questions must be addressed during each of three stages: founding, initial establishment, and rearing for field release (Table 1). Further detail and references on this process can be found in Ref.^[1]. The most difficult task is to evaluate the trade-off between rearing quantity and field quality (Fig. 1), because evaluating field quality is difficult; however, it is very important to the economics of captive rearing that future research provides this information.

REFERENCES

1. Nunney, L. Managing small populations: a population genetic perspective. In *Quality Control and Production of Biological Control Agents: Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI Publishing: Wallingford, UK, 2003; 73–87.
2. Boller, E. Behavioral aspects of mass-rearing of insects. *Entomophaga* **1972**, *17*, 9–25.
3. Pimentel, D. Population dynamics and importance of evolution in successful biological control. In *Handbook of Pest Management*; Pimentel, D., Ed.; CRC Press: Boca Raton, FL, 1990; Vol. 2, 171–175.
4. Bartlett, A.C. Maintaining genetic diversity and laboratory colonies of parasites and predators. In *Applications of Genetics to Arthropods of Biological Control Significance*; Narang, S.K., Bartlett, A.C.,

- Faust, R.M., Eds.; CRC Press: Boca Raton, FL, 1994; 133–145.
5. Burton, R.S.; Rawson, P.D.; Edmands, S. Genetic architecture of physiological phenotypes: empirical evidence for coadapted gene complexes. *Am. Zool.* **1999**, *39*, 451–462.
 6. Leppla, N.C.; Huettel, M.D.; Chambers, D.L.; Ashley, T.R.; Miyashita, D.H.; Wong, T.T.Y.; Harris, E.J. Strategies for colonization and maintenance of the Mediterranean fruit fly. *Entomologia Experimentalis et Applicata* **1983**, *33*, 89–96.
 7. Miyatake, T.; Yamagishi, M. Rapid evolution of larval development time during mass-rearing in the melon fly, *Bactrocera cucurbitae*. *Researches on Population Ecology* **1999**, *41*, 291–297.
 8. McPhee, M.E. Generations in captivity increases behavioral variance: considerations for captive breeding and reintroduction programs. *Biol. Conserv* **2003**, *115*, 71–77.
 9. Roush, R.T.; Hopper, K.R. Use of single family lines to preserve genetic variation in laboratory colonies. *Ann. Entomol. Soc. Am.* **1995**, *88*, 713–717.

Filariasis

N. Pradeep Kumar

Molecular Entomology, Vector Control Research Centre (Indian Council of Medical Research), Pondicherry, India

INTRODUCTION

Human filariasis is caused by a group of infectious nematode parasites belonging to the Order Filariidae. These nematodes cause a variety of clinical diseases in humans, including lymphatic filariasis, onchocerciasis, loiasis, and mansonellosis.

OVERVIEW

The parasites involved in filariasis can be grouped into three categories based on the normal tissues inhabited by the adult worms: 1) *Wuchereria bancrofti*, *Brugia malayi*, and *Brugia timori*—lymphatic system; 2) *Mansonella perstans* and *Mansonella ozzardi*—body cavity; and 3) *Loa loa*, *Onchocerca volvulus*, and *Mansonella streptotraca*—subcutaneous tissues. Species of these parasites are identified by the microfilaria morphology.^[1]

Among these diseases, lymphatic filariasis causes the greatest impact on humans and is globally distributed (in tropical and subtropical regions). However, filariasis caused by *O. volvulus* (onchocerciasis or river blindness) and *L. loa* (Loiasis) are also acute problems that may lead to blindness. These diseases are prevalent in Africa and in South and Central America (loiasis is restricted to tropical Africa). *M. ozzardi*, *M. streptotraca*, and *M. perstans* cause mansonellosis, which is considered a mild illness.

This article will primarily focus on lymphatic filariasis. Loiasis and mansonellosis will be briefly discussed. Onchocerciasis is presented in another article.

LYMPHATIC FILARIASIS

Human lymphatic filariasis, transmitted by mosquitoes (Diptera: Culicidae), can cause massive swelling of legs (elephantiasis) and genitals (hydrocele) in its chronic stages. Also, in some cases, swelling of arms or female mammary glands have been recorded. This debilitating scourge of mankind, second in impact only to leprosy,^[2] causes significant economic loss and social discrimination. More than 1.1 billion people are at risk of infection in 73 countries located in the tropics and

subtropics of Africa, Asia, the Western Pacific and parts of the Americas. Approximately 120 million people are infected.^[2]

W. bancrofti

Almost 90% of lymphatic filariasis cases are caused by *W. bancrofti*.^[2] Generally referred to as “Bancroftian filariasis,” or “urban filariasis,” this disease is chiefly a by-product of unplanned urbanization with poor wastewater disposal. Typical symptoms of filariasis are: 1) microfilaremia; 2) acute manifestations (filarial fever, adenolymphangitis, epididymo-orchitis, inflammatory nodules in the scrotum, breast, and subcutaneous tissues); and 3) chronic manifestations (hydrocele, lymphedema, elephantiasis, chyluria, chronic epididymitis, funiculitis, and lymphedema of vulva). The pathogenicity and symptomatology of the disease vary in different localities.

Three physiological strains of filarial parasite exist (based on the prevalence of microfilaria in the peripheral blood circulation): 1) nocturnally periodic strain; 2) nocturnally subperiodic strain; and 3) diurnally subperiodic strain.^[1] Four ecological types also exist (based on the vector that transmits the disease): *Culex* type, *Anopheles* type, *Aedes* type, and *Mansonia* type.^[1] Genetic variability exists among populations of the nocturnally periodic strain of the species.^[3]

B. malayi and *B. timori*

Two other parasites involved in lymphatic filariasis are *B. malayi* and *B. timori*. The disease caused by these parasites is generally referred to as “Brugian filariasis,” or “rural filariasis,” and is prevalent in Southeast Asian and Western Pacific countries (*B. timori* is restricted to the Lesser Sunda Islands of Indonesia). Nocturnally periodic, nocturnally subperiodic, and non-periodic strains have been reported for *B. malayi*,^[4] while only a nocturnally periodic form has been reported for *B. timori*.^[5] The symptoms of Brugian filariasis are similar to Bancroftian filariasis, with the exception of hydrocele and vulvar lymphedema.

The genomes of filariasis parasites are currently being mapped and the complete sequencing of the

B. malayi genome is ongoing.^[6] Repetitive DNA is a significant component of both *B. malayi* and *W. bancrofti* genome, consisting about 10% of the whole genome (a 322-bp repeat, Hha I) for the former,^[7] and with a lesser copy number (only about 300 copies per haploid genome;^[8] 188-bp repeat, Ssp I) for the latter.^[9]

The diagnosis of filariasis is determined by conventional finger prick and blood smear examination for parasites. The repetitive DNA elements in the genomes of the parasites and other DNA markers are also used for the parasite species identification.^[9–11] Immunological kits such as ICT (immunochromatographic test) are also available for detection of infection.^[12,13]

Vectors

Altogether, 63 species of mosquitoes have been reported as vectors of Bancroftian filariasis worldwide.^[14] The most important vector involved in transmission is *Culex quinquefasciatus*, a ubiquitous mosquito species in the tropics and subtropics, which breeds in stagnant effluent wastewater produced in an urban situation or discarded containers.^[15] Other culicines that are involved in the transmission of filariasis are *Cx. pallens* in China, Japan, and the United States, and *Cx. molestus* in temperate regions. *Anopheles* species such as *An. gambiae*, *An. arabiensis*, *An. meras*, *An. melas*, and *An. funestus* (tropical Africa); *An. culicifacies* (Sri Lanka); *An. sinensis* (China); *An. maculatus* and *An. whartoni* (Malaya); *An. minimus* (Philippines); and *An. punctulatus* group (Papua New Guinea) are all vectors of filariasis in some regions. *Aedes* species such as *A. polynesiensis* (Polynesia, Samoa), *A. niveus* (Andaman Nicobar islands in India), *A. poecilus* (Philippines), *A. vigilax* (New Caledonia), *A. kochi* (Papua New Guinea), *A. tongae*, *A. tabu*, *A. kesseli*, *A. oceanicus* (Tonga), *A. samoanus*, *A. tutuilae*, *A. upotensis* (Samoa), etc., and *Mansonia* species, viz., *Ma. indiana* (Guinea), also act as vectors of this disease.

Brugian filariasis is chiefly transmitted by mosquitoes belonging to *Mansonia* genus (*Ma. annulifera*, *Ma. annulata*, *Ma. uniformis*, *Ma. indiana*, *Ma. dives*, *Ma. bonneae*) and *Coquillettidia* genus (*Cq. crassipes*).^[14] These species of mosquitoes mainly breed in association with aquatic weeds such as *Pistia* sp., *Salvinia* sp., *Eichhornia* sp., *Isachne* sp., etc.^[16] Also, anopheline mosquitoes such as *An. barbirostris*, *An. sinensis*, *An. lesteri*, *An. campestris* and *Aedes* species, viz., *A. togoi*, *A. kianensis*, and *A. kwaiangensis* (China), can act as vectors of Brugian filariasis in different regions. *An. barbirostris* is the only vector of *B. timori*.

Transmission Dynamics

Adult worms of *W. bancrofti*, *B. malayi*, and *B. timori* live in human lymphatic system. The microfilariae produced by the female reach the blood and circulate in the peripheral circulatory system, often synchronized with peak biting activity of the vector. These microfilariae then enter the midgut of vector mosquitoes while they have a blood meal from the infected person. From there, microfilariae penetrate the peritrophic membrane and gut wall of the mosquito. They develop and molt twice in the flight muscles to the L3 form (infective larvae). This stage migrates through the entire body of the mosquito and eventually reaches the proboscis (the mouthparts). When an infected mosquito bites a human, the filaria exit the proboscis and enter the human host through the mosquito-bite wound. The infective larvae then enter into the blood circulation of man. Through the blood stream, the infective larvae find its way to the lymph nodes and molt into the juvenile fourth instars and to the adult worms.^[1]

The extrinsic cycle of incubation of the parasite (in the mosquito host) last for 12–13 days^[1] for *W. bancrofti* and 7–8 days for *B. malayi*.^[17] The number of infective bites required for a patent infection also varies for both *W. bancrofti* and *B. malayi* from one species of mosquito vector to another as well as from region to region.

Very little is known about the development of filaria in man because no successful animal model is available for *W. bancrofti*. In vitro culture of L3 has been accomplished only to the L4 stage for *W. bancrofti*.^[18] In *B. malayi*, in vitro culture has been accomplished to the L4 and adult stages.^[19] The adult worms of *B. malayi* produced live microfilariae in the culture after 75–100 days.^[14]

The prepatent period of infection was recorded to be lesser for *B. malayi* (3.5 months) and *B. timori* (3 months) compared to that of *W. bancrofti* (7 months).^[14] The incubation period of disease ranges from 8 to 16 months or longer for indigenous inhabitants in endemic regions.^[14] When the patient reaches the chronic state of infection, the parasites may be found dead and calcified, blocking the lymphatic system.^[1]

CONTROL OF FILARIASIS

Filariasis control should include: 1) control of vector population (chemical or environmental measures); 2) reduction of parasitemia in human population [by chemotherapeutic drugs such as diethylcarbamazine citrate (DEC), albendazole, or ivermectin]; and 3) proper awareness and prevention training in the community

to encourage active community participation in control programs. The optimal control strategy is one that can be integrated into the community lifestyles. Furthermore, it would be desirable if the adopted control strategies would directly benefit the economic status of the community. However, the adopted strategy should be appropriate to the region, and not directed by a “top-down” system.

The best example for the successful control of filariasis is the one achieved by China (1956–1994).^[20] Diethylcarbamazine chemotherapy was chosen as the intervention measure. Using DEC, 864 counties and cities in 15 regions achieved successful control of filariasis. Surveillance conducted during the past few years demonstrated that the transmission of both Bancroftian and Brugian filariasis has been virtually interrupted in most regions by this program, reaching the criterion for effective control of filariasis.^[20] The notable examples are the control of filariasis in Shandong, Hubei, Fujian province, Guangdong, Kinmen islands, and Guizhou and Henan provinces. The present prospective of the country is the elimination of this disease (current estimate—0.23 million).^[21] In India, despite a nationwide control program, an increase in the prevalence of lymphatic filariasis has been recorded, possibly as a result of a tremendous increase in the human population. The current estimate of filariasis prevalence (microfilaremia and symptomatic cases) is 47.66 million,^[22] the highest figure recorded for filariasis prevalence in any country in the globe.^[21] A successful control program on Brugian filariasis control in Kerala state (the most important endemic region of the disease in India) was recently undertaken, using community-oriented programs involving an Integrated Vector Management Strategy and DEC therapy.^[23] Little information is available on the control programs on filariasis in African countries except that from the United Republic of Tanzania. In the Americas, Brazil reports reemergence of transmission of filariasis.^[1] In 1997, the World Health Assembly adopted a resolution for global elimination of filariasis.^[2] The World Health Organization (WHO) aims to treat more than 1 billion people exposed to the risk of infection with a dose of medicines (DEC and albendazole or albendazole plus ivermectin), theoretically eliminating the disease in approximately 20 years.^[24]

Loiasis

Loiasis, caused by *L. loa*, is a zoonotic disease affecting about 1 million population in the forested areas of Central and West African countries such as Zaire, Cameroon, Gabon, Congo, Nigeria, and Central Africa.^[25] Loiasis is characterized by temporary

swellings in regions (such as limbs) where migrating adult worms occur, mostly on the limbs. These swellings are referred to as “Calabar swelling” and can be crippling. Frequently, adult worms migrate to the conjunctiva of the eyes, but rarely cause permanent ocular damage. Microfilariae of *L. loa* are diurnally periodic in man.^[1] The vector species involved are tabanids (deerflies), *Chrysops dimidiatus* and *C. silaceus*.

Mansonellosis

Mansonellosis is a disease caused by *M. perstans*, *M. streptotraca*, and *M. ozzardi* worms. These are usually nonpathogenic or may cause mild pathogenicity. *M. perstans* is prevalent in Central and West Africa, South America, Mexico, Trinidad, and the Caribbean.^[26] Vectors of *Mansonella* species are midges such as *Culicoides milnei* and *C. grahamii*. *M. streptotraca* transmitted by *C. grahamii* is prevalent in tropical Africa.

M. ozzardi is widespread in South America and the Caribbean, where the microfilaria rate reported from some areas is more than 90% of the population.^[27] Vectors of this disease are *Culicoides furens*, *C. phlebotomus*, and *C. insinuatatus*. *Simulium* species of blackflies may also play a role in the transmission of *M. ozzardi*.

CONCLUSION

Lymphatic filariasis, the major concern among human filarial infections, is presently included under the disease elimination program of World Health Organization and its member nations. The strategy mooted is to liquidate the parasite population in the human host by chemotherapeutical measures. Adopting a concerned effort by different endemic countries and with the help of active community participation in the programs, we could hope the disease will be eliminated from the globe in another two decades, as proposed.

REFERENCES

1. Sasa, M. *Human Filariasis—A Global Survey of Epidemiology and Control*; University of Tokyo Press: Tokyo, 1976.
2. World Health Organization. *World Health Report, 1998—Life in the 21st Century, A Vision for All*. World Health Organization: Geneva, 1998.
3. Pradeep Kumar, N.; Patra, K.P.; Hoti, S.L.; Das, P.K. Genetic variability of the human filarial parasite, *Wuchereria bancrofti*, in south India. *Acta Trop.* **2002**, *82* (1), 67–76.
4. Kanda, T.; Joesoef, A.; Imai, Y.; Suzuki, H.; Yoneyama, K. Microfilarial periodicity analysis of the survey data

- from six localities in Indonesia. Southeast Asian J. Trop. Med. Public Health **1979**, *10* (1), 32–50.
5. Partono, F.; Pribadi, P.W.; Soewarta, A. Epidemiological and clinical features of *Brugia timori* in a newly established village. Karakuak, West Flores, Indonesia. Am. J. Trop. Med. Hyg. **1978**, *27* (5), 910–915.
 6. Blaxter, M.; Aslett, M.; Guiliano, D.; Daub, J. Parasitic helminth genomics. Filarial Genome Project. Parasitology **1999**, *118*, S39–S51(Suppl.).
 7. Lizotte, M.R.; Supali, T.; Partono, F.; Williams, S.A. A polymerase chain reaction assay for the detection of *Brugia malayi* in blood. Am. J. Trop. Med. Hyg. **1994**, *51* (3), 314–321.
 8. Zhong, M.; McCarthy, J.; Bierwert, L.; Lizotte-Waniewski, M.; Chanteau, S.; Nutman, T.B.; Ottesen, E.A.; Williams, S.A. A polymerase chain reaction assay for detection of the parasite *Wuchereria bancrofti* in human blood samples. Am. J. Trop. Med. Hyg. **1996**, *54* (4), 357–363.
 9. Lucena, W.A.; Dhaliya, R.; Abath, F.G.; Nicolas, L.; Regis, L.N.; Furtado, A.F. Diagnosis of *Wuchereria bancrofti* infection by the polymerase chain reaction using urine and day blood samples from microfilaraemic patients. Trans. R. Soc. Trop. Med. Hyg. **1998**, *92* (3), 290–293.
 10. Torres, E.P.; Ramirez, B.L.; Salazar, F.; Pasay, M.C.; Alamares, J.G.; Santiago, M.L.; Hafalla, J.C. Detection of Bancroftian filariasis in human blood samples from Sorsogon province, the Philippines by polymerase chain reaction. Parasitol. Res. August **2001**, *87* (8), 677–679.
 11. McCarthy, J.S.; Zhong, M.; Gopinath, R.; Ottesen, E.A.; Williams, S.A.; Nutman, T.B. Evaluation of a polymerase chain reaction-based assay for diagnosis of *Wuchereria bancrofti* infection. J. Infect. Dis. **1996**, *173*, 1510–1514.
 12. Njenga, S.M.; Wamae, C. Evaluation of ICT filariasis card test using whole capillary blood: Comparison with Knott's concentration and counting chamber methods. J. Parasitol. **2001**, *87*, 1140–1143.
 13. Sunish, I.P.; Rajendran, R.; Satyanarayana, K.; Munirathinam, A.; Gajanana, A. Immunochromatographic test (ICT) for estimation of true prevalence of Bancroftian filariasis in an endemic area in southern India. Trans. R. Soc. Trop. Med. Hyg. **2001**, *95*, 607–609.
 14. World Health Organization. *Lymphatic Filariasis: The Disease and its Control, Fifth Report of the WHO Expert Committee on Filariasis*. WHO Technical Report Series No. 821; WHO: Geneva, 1992.
 15. Rajagopalan, P.K.; Das, P.K.; Subramanian, S.; Vanamail, P.; Ramaiah, K.D. Bancroftian filariasis in Pondicherry, south India: I. Pre-control epidemiological observations. Epidemiol. Infect. **1989**, *103* (3), 685–692.
 16. Sabesan, S.; Rajendran, G.; Pradeep Kumar, N. Bionomics of mansonioides mosquitoes in relation to community structure of hydrophytes/breeding habitats in Cherthala, Kerala. Indian J. Clin. Biochem. **1997**, *12*, 36–38 (Suppl.).
 17. Pradeep Kumar, N.; Sabesan, S.; Panicker, K.N. Susceptibility status of *Mansonia annulifera* to *Brugia malayi* parasites in Cherthala, Alappuzha district, Kerala. Indian J. Exp. Biol. **1998**, *36* (8), 829–831.
 18. Franke, E.D.; Riberu, W.; Wiady, I. In vitro cultivation of third stage larvae of *Wuchereria bancrofti* to the fourth stage. Am. J. Trop. Med. Hyg. **1987**, *37* (2), 370–375.
 19. Riberu, W.A.; Atmosoedjono, S.; Purnomo; Tirtokusumo, S.; Bangs, M.J.; Baird, J.K. Cultivation of sexually mature *Brugia malayi* in vitro. Am. J. Trop. Med. Hyg. **1990**, *43* (1), 3–5.
 20. Anonymous. A great success in lymphatic filariasis control in China. National Technical Steering Group for Filariasis Control and Research, MOPH. Chung Kuo Chi Sheng Chung Hsueh Yu Chi Sheng Chung Ping Tsa Chih **1995**, *13* (2), 81–85.
 21. World Health Organization. *World Health Report, 1999—Making a Difference*. World Health Organization: Geneva, 1999.
 22. Sabesan, S.; Palaniyandi, M.; Das, P.K. Mapping of lymphatic filariasis in India. Ann. Trop. Med. Hyg. **2000**, *94* (6), 591–606.
 23. Panicker, K.N.; Dhanda, V. Community participation in the control of filariasis. World Health Forum **1992**, *13* (2–3), 177–181.
 24. Das, P.K.; Ramaiah, K.D.; Augustin, D.J.; Kumar, A. Towards elimination of lymphatic filariasis in India. Trends Parasitol. **2001**, *17* (10), 457–460.
 25. Rodhain, F. Hypotheses on the dynamic ecology of Loa infections. Bull. Soc. Pathol. Exot. Filiales **1980**, *73* (2), 182–191.
 26. Ufomadu, G.O.; Nwoke, B.E.; Akoh, J.I.; Sato, Y.; Ekejindu, G.O.; Uchida, A.; Shiwaku, K.; Tumbau, M.; Ugomo, K.K. The occurrence of loiasis, mansonellosis and wuchereriosis in the Jarawa River Valley, central Nigeria. Acta Trop. **1990**, *48* (2), 137–147.
 27. Lowrie, R.C., Jr.; Raccurt, C. *Mansonella ozzardi* in Haiti II. Arthropod vector studies. Am. J. Trop. Med. Hyg. **1981**, *30* (3), 598–603.

Fire Ant Attacks on Humans and Animals

Jerome Goddard

Mississippi Department of Health, Jackson, Mississippi, U.S.A.

Richard de Shazo

Department of Medicine, University of Mississippi Medical Center, Jackson, Mississippi, U.S.A.

INTRODUCTION

Imported fire ants were introduced from South America into the southern United States approximately 80 years ago and have been spreading ever since. There are actually two species involved—*Solenopsis richteri* and *Solenopsis invicta*—and a hybrid sometimes called *S. invicta* × *richteri*. The red imported fire ant, *S. invicta*, is the most abundant and widespread of the two species, infesting over 300 million acres. The ants aggressively sting intruders disturbing their mounds or feeding trails and have been known to impact both humans and animals in a number of ways. There are medical effects (allergic reactions and skin infections), nuisance effects (uncomfortable stings and invasion of food products or homes), and economic effects (structural and equipment damage and damage to lawns or gardens).^[1] In animals, fire ant attacks may result in blindness, reduced weight gain, or death. Indirectly, fire ants may affect wildlife by reducing food sources in nature such as invertebrates.

ATTACKS ON PEOPLE

Fire ants have expanded their habitat and density from the Gulf South east to Virginia and west to California, resulting in massive sting attacks of individuals (Fig. 1). These attacks have occurred primarily in private homes or health-care facilities.^[2–7] A 5-day-old infant stung at home had a near fatal response, while a 26-month-old toddler developed corneal opacities.^[5,6] The death of a 3-month-old infant subsequent to numerous fire ant stings in an upscale subdivision of Phoenix, Arizona was recently reported in the lay press (The Arizona Republic—May 20, 2003). At least two healthy adults and one patient with Alzheimer's disease stung at home have survived fire ant attacks without sequelae. Recently, an anaphylactic reaction was reported in a nursing home patient after a fire ant attack.^[8]

Our experience suggests that residents at health-care facilities in fire ant endemic areas are especially at risk for fire ant attacks for several reasons. Common

factors in stinging events include fire ant infestation of a facility and an immobile or cognitively impaired patient. When fire ants are noted in a health-care facility, health-care personnel are often unaware of the behavior of these insects and the special measures required for their control. Sometimes pesticides ordinarily used to control indoor pests do not control fire ant infestations as they do not kill the fertile queen that is located outdoors in the soil.^[4] Service contracts with pest control companies may be ambiguous about both company and facility responsibilities. Moreover, these contracts sometimes fail to provide for pest control out-of-doors, the natural habitat of fire ants. In many cases where sting attacks have occurred, fire ant colonies have been found in large numbers on the grounds of facilities and even adjacent to the perimeter of the facility slab. Once fire ants are detected in a facility, facility personnel often have no formal procedures to report and expeditiously eliminate infestation. Spraying worker ants with insect sprays may kill a few workers while the queen continues to produce replacements. Usually, poison baits are required to eliminate the queen and colony.

Contributing to indoor stinging events, ant colonies may move closer to or into occupied buildings under special circumstances such as drought, flooding, cold, or high density of colonies.^[9] In such cases, colonies may be located adjacent to or under foundation slabs or in outer building walls. In the case of medical facilities, worker ants may explore patient rooms and patients themselves, looking for food. Imported fire ants ingest, among other things, sugars, some amino acids, and oils containing polyunsaturated fats in liquid form. Thus it is no surprise that mucous membranes of the mouth, nose, and eyes have been sought out by ants in attacks of humans.^[2,9] When disturbed in their feeding process by movement or vibration, ants on or in close contact with patients may sting these individuals multiple times.

Because nothing seems able to stop the geographic expansion of imported fire ants—except perhaps severe cold weather—we have published recommendations for prevention and management of fire ant infestation in health-care facilities and the emergency



Fig. 1 Fire ant stinging human thumb (photo courtesy of Dr. James Jarratt, Mississippi Cooperative Extension Service, Mississippi State University, with permission).

management of massive fire ants stings.^[10] In addition to native infestations in South America and the expanding habitat in North America and Puerto Rico, fire ant colonies have recently been detected in Australia.^[11] Personnel in health-care facilities in endemic areas must become aware of the risk to patients posed by these insects and various ways to control infestations. Physicians and pest control personnel need to participate in this overall process. Unless this occurs, we anticipate that attacks of individuals in health-care facilities will be a continuing problem in the United States and elsewhere.

ATTACKS ON ANIMALS

Pets, domestic animals, and wildlife are also impacted by fire ants.^[1] Newborn or hatchling animals may be susceptible to attack because they are unable to escape. Because the ants are attracted to mucous areas, animals are often stung around the eyes, leading to blindness, or around the mouth and throat, leading to suffocation. A confined animal is especially at risk. One study of cotton rats captured in Sherman traps (checked after 6 hr) revealed that 19% of rats captured were attacked by ants—20 were dead and covered with ants; 13 were alive and covered with ants, partially eaten alive.^[12]

The effect of fire ants on young wild animals remains mostly unknown. There have been a few reports of fire ant predation on young birds. Mrazek^[13] observed fire ants killing nestling black skimmers (*Rynchops niger*) and gull-billed terns (*Sterna nilotica*) and adult birds abandoning nests invaded by fire ants.

Sikes and Arnold^[14] observed red imported fire ants attacking live cliff swallow nestlings in 212 of 357 nests. The death of large numbers of bluegill sunfish in Mississippi and Alabama has been attributed to ingestion of fire ants.^[15]

REFERENCES

1. Vinson, S.B. Invasion of the red imported fire ant. *Am. Entomol.* **1997**, *43*, 23–39. Spring issue.
2. deShazo, R.D.; Banks, W.A. Medical consequences of multiple fire ant stings occurring indoors. *J. Allergy Clin. Immunol.* **1994**, *93*, 847–850.
3. deShazo, R.D.; Williams, D.F. Multiple fire ant stings indoors. *S. Med. J.* **1995**, *88*, 712–715.
4. deShazo, R.D.; Williams, D.F.; Moak, E.S. Fire ant attacks on residents in health care facilities: a report of two cases. *Ann. Intern. Med.* **1999**, *131*, 424–429.
5. Amador, M.; Busse, F.J., Jr. Corneal injury caused by imported fire ants in a child with neurologic compromise. *J. Pediatr. Ophthalmol. Strabismus* **1998**, *35*, 55–57.
6. Hardwick, W.E.; Royall, J.A.; Pettit, B.A.; Tilden, S.J. Near fatal fire ant envenomation of a newborn. *Pediatrics* **1992**, *90*, 662–664.
7. Diaz, J.D.; Lockey, R.F.; Stablein, J.J.; Mines, H.K. Multiple stings by imported fire ants (*Solenopsis invicta*) without systemic results. *S. Med. J.* **1989**, *82*, 775–777.
8. deShazo, R.D.; deShazo, M.D.; Goddard, J. Fire ant attacks on patients in nursing homes: an increasing problem. *Am. J. Med.* **2004**, *116*, 843–846.
9. Porter, S.D.; Fowler, H.G.; MacKay, W.P. Fire ant mound densities in the United States and Brazil (Hymenoptera: Formicidae). *J. Econ. Entomol.* **1992**, *85*, 1154–1161.
10. Goddard, J.; Jarratt, J.; deShazo, R.D. Recommendations for prevention and management of fire ant infestations in healthcare facilities. *S. Med. J.* **2002**, *95*, 627–633.
11. Solley, G.O.; Vanderwoude, C.; Knight, G.K. Anaphylaxis due to red imported fire ant sting. *Med. J. Aust.* **2002**, *176*, 518–519.
12. Flickinger, E.L. Observations of predation by red imported fire ants on live-trapped wild cotton rats. *Texas J. Sci.* **1989**, *41*, 223–224.
13. Mrazek, R.W. The Relationship of the Fire Ant to the Young of Birds Nesting on Two Spoil Islands in the Laguna Madre. MS thesis, Texas A&I University: Kingsville, 1974. Unpublished, 122 pp.
14. Sikes, P.J.; Arnold, K.A. Red imported fire ant predation on cliff swallow nestlings in east-central Texas. *Southwest. Nat.* **1987**, *31*, 105–106.
15. Harwood, R.F.; James, M.T. *Entomology in Human and Animal Health*, 7th Ed.; Macmillan Publishing Co.: New York, 1979.

Flooding: Physiological Adaptations and Weed Control

Rudraraju A. Raju

Agricultural Research Station, A.R. Andhra Pradesh Agricultural University (AR APAU),
Maruteru, Hyderabad, India

INTRODUCTION

Flooding kills weeds by excluding air from their environment. Most of the terrestrial weeds are highly susceptible to stagnant water. Flooding is a common crop husbandry method of controlling young (one to four leaf stage) mesophytic weeds in rice fields the world over. Rice, the major food crop, is cultivated in 112 countries of Asia, Africa, and the American continents. The annual world production is about 372 million tons, gleaned from about 143 million hectares. Throughout the world, flooding is practiced in 45.8 million hectares to control weeds emerging from paddies. About 350 species of weeds infest rice fields and, among them, 18 species are found to be the most pernicious. Worldwide, weeds cause an estimated loss of 74 million tons of rice every year. In addition to yield and quality losses, there are financial losses as a result of the cost of herbicides and the mechanical operations required to control weeds. It is estimated that the practice of flooding saves nearly 55.8 million tons of rice each year by suppressing the weed competition.

Flooding brings about a series of physicochemical, electrochemical, and biochemical changes in soil-plant relationships, completely different from normal aerated soils. Because of the demand for oxygen in the soil for biological activity and the slow renewal rate through the flood water, oxygen in warm soils containing an energy source is usually depleted by soil biota within a day or so after flooding. Biological reduction processes result in the accumulation of reduced chemical ions (such as iron and manganese), products of anaerobic reduction (such as methane, hydrogen, carbon dioxide, and nitrogen), and organic acids (such as lactic acid, malic acid, and acetic acid), which are harmful to plant roots.

Injurious effects on plants are caused by several metabolic imbalances ultimately resulting from insufficient oxygen. The transport of cytokinin hormones is retarded from leaves and stems to the root tips. The insufficient absorption of minerals, accompanied by slower photosynthesis and carbohydrate translocation, reduces the root permeability to water because of insufficient oxygen. The supply of adenosine 5'-triphosphate (ATP) is limited because the electron

transport system and the Krebs's cycle cannot function without oxygen. Under hypoxic and anoxic conditions, the inhibition of respiration and metabolism takes place in all parts of the citric acid cycle in roots. Glycolysis occurs by the lysis of sugar, resulting in the breakdown of sugar to ethanol. No upland weed is tolerant to high ethanol concentrations in cells. Weed growth in different soil water regimes depends on various adaptive characters (Table 1).

FLOODING SYSTEMS IN RICE

Dynamic Flooding

Continuous flowing irrigation is practiced in intermountain areas, terraces of mountain valleys, and hilly regions in India, Philippines, Japan, and the Republic of Korea. The surplus of water coming from mountain tops is used for irrigation and weed control in rice in these areas. The terraces of the Himalayas in Kashmir, the Dehradun and Assam valleys in India, and the Hokkaido and Tohoku areas of central Japan are the best examples of this type of rice cultivation through flooding. Although flow flooding controls the weeds to some extent, the crop is not free from weed competition. The growth of certain weeds such as *Echinochloa colona*, *Eragrostis japonica*, and *Marsilea quadrifolia* is encouraged as the water contains abundant oxygen. This practice requires 4500–5000 mm of rainfall per season.

Static Flooding

Continuous static flooding is usually practiced in most south Asian, Latin American, and African regions where adequate water supplies are available. The depth of flooding varies from 5 to 15 cm in depth, and rainfall requirement ranges from 600 to 1500 mm per crop season. Certain grassy weeds are effectively controlled in shallow water regime, but broad leaf and sedge weeds proliferate as usual as a result of certain adaptations. Certain semiaquatic weeds such as *Sphenoclea zeylanica*, *Ipomoea aquatica*, and *Monochoria vaginalis* can also pose competition to the rice crop.

Table 1 Weeds that flourish in various hydroecosystems

| Scientific name | Common name | Family | Life cycle | Mode of reproduction |
|--|--------------------------|----------------|-------------------------|-----------------------------|
| <i>Well-aerated soils</i> | | | | |
| <i>Amaranthus spinosus</i> L. | Spiny amaranth | Amaranthaceae | annual | small seed |
| <i>Ageratum conyzoides</i> L. | Poultry lice plant | Asteraceae | annual | slender achene |
| <i>Eclipta prostrata</i> (L.) L. | Eclipta | Asteraceae | annual | achene |
| <i>Cyperus rotundus</i> L. | Purple nut sedge | Cyperaceae | perennial | tubers, seeds |
| <i>Cynodon dactylon</i> (L.) pers | Bahama grass | Poaceae | perennial | runners, seed |
| <i>Dactyloctenium aegyptium</i> (L.) Beccut | Crow foot grass | Poaceae | annual | tiny seed |
| <i>Digitaria ciliaris</i> (Retz) Koel | Crab grass | Poaceae | annual | narrow caryopsis |
| <i>Eleusine indica</i> (L.) Gaertn | Goose grass | Poaceae | annual | oblong caryopsis |
| <i>Ischaemum rugosum</i> Salisb | Rugose grass | Poaceae | straggling annual | point-tipped seed |
| <i>Leersia hexandra</i> SW | Leersia grass | Poaceae | perennial | root stocks, seed |
| <i>Paspalum scrobiculatum</i> L. | Paspalum grass | Poaceae | perennial | stem branches, seed |
| <i>Saturation regime</i> | | | | |
| <i>Brachiaria mutica</i> (Forsk) Stapf | Buffalo grass | Poaceae | stout perennial | runners, seed |
| <i>Commelina benghalensis</i> L. | Fire leaf | Commelinaceae | prostrate annual | seed, underground stems |
| <i>Echinochloa colona</i> (L.) Link | Barnyard grass | Poaceae | tufted annual | seed |
| <i>Fimbristylis littoralis</i> Gaud | Devil sedge | Cyperaceae | tufted annual | tiny seed |
| <i>Shallow-flooded regime</i> | | | | |
| <i>Alternanthera sessilis</i> (L.) RBr | Water amaranth | Amaranthaceae | prostrate annual | seed, vegetative branches |
| <i>Cyperas difformis</i> L. | Brahmin sedge | Cyperaceae | tufted annual | seed |
| <i>Cyperas iria</i> L. | Unubal sedge | Cyperaceae | tufted annual | triangular achene |
| <i>Echinochloa glabrescens</i> Munro ex Hook | Pyramidal barnyard grass | Poaceae | stout annual | seed |
| <i>Leptochloa chinensis</i> (L.) Nees | Sprangle top | Poaceae | stout annual | seed |
| <i>Leptochloa filiformis</i> L. | Feather grass | Poaceae | slender annual | seed |
| <i>Monochoria vaginalis</i> (Burms Prege) | Mono Choria | Pontederiaceae | broadleaf annual | tiny seed |
| <i>High pond potential</i> | | | | |
| <i>Pistia stratiotes</i> L. | Pistia | Araceae | free-floating perennial | offshoots, seed |
| <i>Ipomoea aquatica</i> Forsk | Water Ipomea | Convolvulaceae | spreading perennial | seed and vegetable cuttings |
| <i>Sphenoclea zeylanica</i> Gaertn | Goose weed | Sphenocleaceae | flesh, hollow annual | tiny seed |
| <i>Ludwigia octovalvis</i> (Jacq) Raven | Lowland clove | Onagraceae | shrubby annual | seed |
| <i>Eichhornia carssipis</i> | Water hyacinth | Pontederiaceae | free-floating perennial | offshoots, seed |

Cyclical Submergence or Intermittent Flooding

Cyclical flooding is practiced in areas where water supplies are limited. Water for flooding is produced at regular intervals. At times, the field is without standing water between two irrigation events, but the soil remains wet enough to avoid water stress. This rotational flooding is highly effective in Taiwan, the Philippines, India, and parts of Latin America. Usually, the system is designed to flood thousands of hectares of rice at 5-day intervals. It is difficult to manage the weeds in the system because of aerobic soil conditions between two irrigations. The spectrum of

weed flora and biomass production of weeds is high compared to static flooding (Table 1).

PHYSIOLOGICAL ADAPTATIONS TO FLOODING

Germination of Weed Seeds Under Flooded Conditions

Certain weeds such as *M. vaginalis* and *E. glabrescens* can germinate in marshy soils. These seeds can also germinate under as much as 10 cm of water because

they require less free oxygen for germination than some mesophytic weed seeds.

An *Echinochloa* spp. seed is capable of releasing oxygen by an enzymatic process that occurs during germination. These seedlings can remain alive under water for nearly 50 days. Any soil cover under water reduces oxygen supply to the seed, decreasing germination. Anaerobic respiration enables these weed seeds to germinate even under low oxygen. Seedlings of *Lep-tochloa chinensis* exhibit a strong hexose utilization mechanism and a highly functional fermentation system, which partially explain why some of their wild rice (*Oryza nivara*) and *Echinochloa cruss-galli* seeds will germinate under hypoxia or anoxia, but they do so by upward protrusion of the radicle. Roots hardly develop, but the coleoptile continues to grow under hypoxia faster than under anoxia. The enhanced coleoptile growth results from the accumulation of ethylene gas by young seedlings, and this plant hormone promotes coleoptile elongation at the expense of root growth.

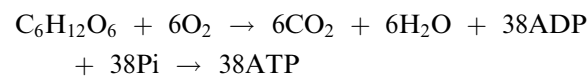
TOLERANCE MECHANISM OF WEEDS TO FLOODED SOILS

Certain weeds such as *Echinochloa glabrescens* and *Marsilea minuta* have physiological adaptations allowing them to survive in waterlogged soils. These plants have a fermentation system that functions to compensate for the decreased aerobic respiration, a factor responsible for the ability of aquatic weeds to germinate and to grow under very low oxygen concentrations. Another defensive mechanism of aquatic weeds under conditions of oxygen deficiency is root lignification, which tends to exclude reduced toxic substances. Waterlogging enhances manganese toxicity. These weeds are more tolerant to high manganese content than mesophytic weeds.

The liberation of energy by hydrophytic weed roots is carried out through anaerobic respiration instead of aerobic metabolism, as in other mesophytic weeds. The phenomenon by which more sugar is issued and more CO₂ and ethanol are produced under anaerobic conditions is called *Pasteur effect*. The Pasteur effect undoubtedly causes decreased carbohydrate reserves of plants in flooded soils and probably helps explain why weeds with swollen rhizomes or thick roots that store carbohydrate reserves can survive anoxia longer.

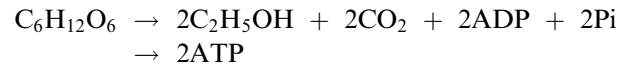
Anaerobic metabolism may result in the production of two molecules of glucose instead of the 686 kcal of energy and 6 mol of CO₂ yielded in anaerobic metabolism.

Aerobic respiration in mesophytic weeds



Because IATP = 12 kcal of energy, 465 cal is released per mole of glucose. Therefore $38 \times 12 = 465$ kcal.

Anaerobic respiration in hydrophytic weeds



Therefore $2 \times 38 = 78$ kcal.

If we equate CO₂ evolution rates in both cases, glucose utilization is 200% greater in hydrophytic weeds and is only 24% as much in fermentation as in respiration.

PECULIAR FEATURES OF SUBMERGED WEEDS

The physiology of submerged weeds present in deep-water rice fields is highly interesting and curious and differs from terrestrial weeds. The submerged plants use oxygen and carbon dioxide most economically. These gases are present in minute quantities in water and 30 times less than atmospheric air. The submerged weeds have an extensive development of a system of air chambers and air spaces in their tissues so that the entire plant body can obtain enough oxygen for respiration by internal circulation. Besides, oxygen produced during photosynthesis is stored in specially designed lacunate tissues, which are distributed in all plant parts. The submerged weeds utilize the dissolved CO₂ and bicarbonates for carbon assimilation mediated by the enzyme carbonyl anhydrase.

Light transmission in water is blocked by various dissolved and suspended constituents. Water molecules absorb ultraviolet and infrared wavelengths of sunlight, while the organic solutes filter the blue and violet parts of the light spectrum. Chlorenchymatous tissues present in mesophyll help in the absorption of diffused light present in water.

VENTILATION OF WATER WEEDS

The absence of oxygen around the roots of hydrophytes inhibits aerobic respiration in roots and allows potentially harmful materials to accumulate in the *rhizosphere*. Certain aquatic weeds such as *S. zeylanica*, *M. vaginalis*, and *Ludwigia octovalvis* develop internal gas spaces (lacunae) in this environment, serving primarily to transport oxygen to buried roots and rhizomes. Oxygen is diffused to the roots from photosynthesizing leaves, and carbon dioxide is diffused from the roots toward the leaves, while the methane produced in sediments during the night circulates throughout the plant. During the day, methane appears

in petioles of older leaves, but not in young leaves. The lacunar system between young and older leaves through which gases move freely by diffusion is continuous.

SURVIVAL OF RICE IN SUBMERGED SOILS

Rice (*O. sativa* L.) is unique among cereals because of its ability to germinate and grow successfully in water-submerged soils. In swamp rice, the fields are inundated throughout the lifespan of the crop. There are some unique anatomical characteristics of rice which enable it to survive in waterlogged soils. Rice roots have spongy cells inside, and these roots are filled with air. The porosity of rice roots increases with the depth of submergence. Oxygen is diffused from the leaves via the tillers and stems to the roots through intercellular spaces or channels in the cortex tissues. It is not known how oxygen reaches young tissues, which are usually devoid of intercellular space. Nevertheless, the mechanism is sufficient not only to meet the oxygen requirement of respiring root cells but also to secrete oxygen or oxidized compounds into the external root microenvironment and to ward off the entry of reduced substances.

As a result of the exudation of oxidized compounds, rice roots become coated with yellowish red precipitates of unknown composition. It is presumed that they are formed by iron and manganese oxides and hydroxides. Root coats are not formed around root tips.

OXIDATION POWER OF ROOTS—THE INDEX OF TOLERANCE

Several field crops have the capacity to oxidize their root environment (*rhizosphere*) when flooded. The root oxidation capacities of common crops, in terms of the amount of naphthylamine oxidized per gram of root during flooding (Table 2), suggest that rice roots have a higher oxidation potential than any other crop. Further, this allows us to understand why sorghum tolerates temporary flooding better than maize.

Another physiological adaptation to flooding is the conversion of acetate, which is produced in the roots from acetyl COA into CO₂ via the glycolic acid cycle, with the concomitant release of O₂ decomposed by a catalase with the production of O₂, which becomes available to the roots. Rice roots thus get oxygen not only through the internal ventilation system but also chemically through enzymatic process.

WEED CONTROL THROUGH FLOODING

Some of the pernicious weeds such as Tiger grass (*Saccharum spontaneum* L.), Johnson grass (*Sorghum*

Table 2 Relative root oxidation power of some field crops under flooded condition

| Crop | Naphthylamine oxidized in 48 hr (mg/g dry root) |
|------------------------------------|---|
| Maize (<i>Zea mays</i>) | 1.4 |
| Oats (<i>Avena sativa</i>) | 2.9 |
| Sorghum (<i>Sorghum bicolor</i>) | 4.0 |
| Wheat (<i>Triticum aestivum</i>) | 4.9 |
| Soybean (<i>Glycine max</i>) | 7.1 |
| Rice (<i>Oryza sativa</i>) | 15.30 |

halepense L.), Purple nutsedge (*Cyperus rotundus* L.), and Thatch grass (*Imperata cylindrica*), which reproduce rapidly by means of highly viable seeds and rhizomes, are very difficult to control. Manual weeding is time-consuming and requires special crowbars for soil digging and collection of rhizomes. Chemical control is often not effective and repeated applications are required.

For these perennials, prolonged flooding for about 2–3 months during the rainy season effectively eradicates these weeds. In India, Tiger grass infests about 3 million hectares of cropped lands. Control measures during winter are not effective because the grass goes under dormancy during the winter. In central India, where irrigation sources are plentiful, the weed is controlled in infested fields by deep ploughing during summer and by impounding plots with water up to 20 cm in depth for 1 week. Then a tooth-pegged beam is run in wet fields to extract Tiger grass rhizomes loosened by flood water. This is followed by wetland rice, which kills the rest of the rhizomes as a result of anaerobiosis.

FLOOD FALLOW

In some parts of coastal India and central India, the fields are deep-flooded for about 4 months during the rainy season to control the heavy infestation of problematic weeds such as bermuda grass (*Cynodon dactylon*), lantana (*Lantana camera*), foxtail (*Setaria glauca*), and *Physalis angulata*. After the rainy season (June to October), the water is released from the fields to sow winter crops such as wheat and chick peas. This practice is called the *Haveli* system, which is very effective in eradicating difficult-to-control weeds. On the eastern coast of India, quack grass (*Angropyron repens*), one of the top 10 weeds of the world, heavily infests upland cropping areas. The weeds propagate and colonize new areas rapidly through rhizomes. These rhizomes grow fast in all directions within a short time and chemical control is not effective. The oldest, yet effective, method of controlling weeds is

through flooding. In summer, soil levees are made and water inundates these fields during the rainy season. The rainwater may reach a depth of up to 1 m in these fields. The weeds, along with fleshy rhizomes, completely decompose as a result of an anaerobic environment that prevails for 2–3 months. This is the surest way of controlling this perennial weed.

CONCLUSION

Flooding of rice paddies has proven highly effective in controlling some weeds in rice production for thousands of years and flooding is still practiced on 46 million hectares in the world. Flooding kills weeds by excluding air from the weed environment. Weed control through flooding has proved highly effective against several pernicious weeds, including tiger grass, Johnson grass, purple nutsedge, and thatch grass.

BIBLIOGRAPHY

- Raju, R.A. *Ecology of Aquatic Weeds and Their Control*; Kalyani Publishers: Ludhiana, 1996; 168 pp.
- Raju, R.A. *Glimpses of Rice Technology*; Agrobios Publishers: Jodhpur, India, 2000; 211 pp.
- Raju, R.A. *Prevalent Weed Flora in Peninsular India*; Allied Publishers PVT LTD: New Delhi, 1998; 271 pp.
- Raju, R.A.; Kona, G.R.S.S. How rice plant survives in submerged soils. *Sci. Report.* **1979**, *16* (4), 271–273.
- Raju, R.A.; Reddy, M.N.; Kondap, S.M. Weed flora as influenced by various hydro-ecosystems. *Indian J. Weed Sci.* **1995**, *27* (3–4), 219–221.
- Raju, R.A.; Reddy, M.N.; Reddy, K.A. Phasic water management for rice in Godavari alluvials. *Indian J. Agron.* **1992**, *37* (1), 26–29.
- Reddy, M.N.; Raju, R.A. Studies on water management of rice on vertisols. *Indian J. Agron.* **1987**, *32* (3), 232–234.
- Rumpho, M.E.; Kennedy, R.A. Anaerobiosis in *Echinochloa crus-galli* seedlings. *Plant Physiol.* **1983**, *72*, 44–49.

Fruit Crop Pest Management: Weeds

Sylvia Guidoni
Alessandra Ferrandino

Dipartimento di Colture Arboree, Università di Torino, Grugliasco, Torino, Italy

INTRODUCTION

Weeds cause an estimated 12% crop loss worldwide; fruit crop loss is estimated to be about 4–5%. Weeds can interfere with fruit crop cultivation and production, competing for water, nutritional elements, light, and air that sometimes are available in reduced quantities. Woody perennial crop system trees or vines might tolerate higher weed populations than agronomic crops. Weeds can produce chemical compounds that can exert allelopathy, resulting in tree growth limitation, leaf fall, or chlorosis. Weeds can also cause indirect damage by creating microclimate conditions that can favor the growth of microorganisms (i.e., fungi and bacteria) and the development of insects and mites that can be carriers of viruses and/or mycoplasmas.

The type of weeds in fruit crop is tied to environmental conditions and to the fruit orchard age. In the first years of the orchard establishment, many different species can develop but as time goes by, flora association tends to become specialized. Perennial gramineae and dicotyledons with main roots or stolon roots and with a great aerial development generally become extensively diffused in the orchard.

Among annual weeds we find many gramineae (*Avena*, *Alopecurus*, *Lolium*, *Poa*, *Bromus*, *Echinochloa*, *Setaria*, *Digitaria*, *Sorghum*) and many dicotyledons (*Stellaria*, *Papaver*, *Lamium*, *Sonchus*, *Senecio*, *Matricaria*, *Polygonum*, *Chenopodium*, *Solanum*, *Amaranthus*, *Portulaca*, *Mercurialis*, etc.). Biennial weeds propagate by seed and their cycle is accomplished in 2 years (*Plantago*, *Taraxacum*, *Geranium*, etc.). Perennial species can propagate by seed, rhizome, root, stolon, or bulb; among the most diffused species, we find *Agropyron*, *Cynodon*, *Rumex*, *Cirsium*, *Allium*, *Equisetum*, *Artemisia*, *Convolvulus*, etc.

In cool climate zones, weed vegetative growth is greatly reduced during extreme seasons because of low or high temperatures and drought (at zero-irrigation zones) while in cool and wet seasons weed development is maximum.

Weed control requires regular field monitoring to assess weed diffusion, botanical characteristics, and vegetative cycle growth, and propagating ways to select the most appropriate strategy of control. Weed control

can be pursued by direct and/or indirect methods. Indirect methods are preventive ways for limiting or avoiding weed development (Table 1). Direct methods include physical, agronomic, chemical, and biological methods. Physical, agronomic, and biological methods have been developed as a result of increasing demands for reduced chemical use and environmental impact.

PHYSICAL METHODS

Fire

In the past, fire was used to control weed development, which led to destruction of vegetation and recovery of clean land for cultivation; nevertheless, it has been demonstrated that a reduced percentage of weed seeds is killed by fire and during the following cultivation cycle weeds can consistently develop.

AGRONOMIC METHODS

Mechanical Tillage

Mechanical tillage implies hoeing, grubbing, and digging; for fruit crops, it is important to operate to a depth not exceeding 10–15 cm to avoid tree root damage. To limit weeds efficaciously, it is necessary to make tillage frequently during spring and summer, when weed development is more intense, and to make deeper tillage during autumn. Tillage number per year depends on environmental conditions (i.e., soil and climate) and weed species.

Tillage carries some disadvantages; it contributes to damage in the soil structure, caused by an excessive sod mincing, which hinders machines from entering the orchard in specific conditions (e.g., after rainfall), particularly in clay soils. Frequent tillage can speed up the organic matter mineralizing process, thereby exhausting soil resources. In slope-tilled orchards, erosion can reach extreme levels.

Tillage shows numerous advantages; it increases tree water efficiency by increasing soil oxygen content that favors soil water capillary lift; it eliminates

Table 1 Indirect methods against weed development in fruit crops

| Aim | Methods |
|--|---|
| Limiting or avoiding the introduction of weeds in the cultivation area | Use manure with no vital weed seeds Accurately clean machines before entering a new field |
| Limiting or avoiding present weed seeds development | Cut down meadows before weed dissemination Soon till sod before weed seeds mature |
| Choosing suitable working techniques | Use the minimum tillage, if possible, so that a reduced percentage of weed seeds comes to surface and can germinate Avoid the use of rotary tillers where rhizome weeds are diffused Work the soil soon in summer to induce weed resistance organ death |
| Rectifying soil characteristics | Neutralize acidity against acidophilus weeds Eliminate stagnation of water as a measure against weeds that can grow in anoxic conditions |
| Increasing fruit tree competitiveness | Do not put manure within rows but only near fruit trees so as not to favor weed development Choose well the moment of implantation as to support the fruit tree rapid development |

nutritional and water competition with weeds; and it increases rainfall water accumulation.

The use of this orchard management strategy is useful in various situations: 1) in level surfaces where erosion is not a real problem; 2) in areas where water is scarce or very expensive; and 3) in the first years of the orchard establishment, to avoid weed competitiveness with young plants.

Mulching

Mulching is applied to cover tree rows during the first years of orchard establishment; organic matter (straw, sawdust, leaves, branch tendrils, and compost), inorganic matter (stones or sands), or plastic films (polyethylene (PE), polyvinyl chloride (PVC)) can be used as cover materials. The use of compost mulch requires high-quality compost. Black-film mulch can increase soil temperatures up to 80°C during summer while the blue-film mulch can contain the temperature increase.

Mulching can offer many advantages (Table 2).

Cover Crops

Despite the possibility that it may reduce crop yield, the use of groundcover vegetation to limit weed development in woody perennial crop systems relies on the fact that, unlike most herbaceous crops, in fruit trees weed competition for sunlight and for nutrient supply is minimal.

Groundcover vegetation strategies imply total or partial association of grass with fruit crops, thus limiting weed development. Cover crops can contribute to regulation of tree development, reducing shoot

development and their growth persistence; they can stimulate fruit tree root development by increasing their water assimilative capacity, which can improve accumulation processes in fruits and reserve organs.

Cover crops play an important role in improving the physical and chemical characteristics of soil. They improve the soil structure, permeability, and porosity, accommodate machines passing on the field, and protect soil from erosion and traffic compaction. They increase the decomposed organic matter content of soil in superficial layers, improving some nutrient availability and uptake.

Cover crops can be natural or artificial, and temporary (adventitious cover crops) or permanent; species of spontaneous grass cover are well adapted to climatic and soil conditions, but they often show a great competitiveness with the cultivated fruit crop. The choice of cover crops for artificial grass covering is a delicate intervention because they require reduced water, nutrition, and light; they should also not

Table 2 Main advantages with mulching

1. Weed elimination
2. Soil structure preservation due to reduced atmospheric events incidence
3. Higher percentage of success at tree establishment
4. Higher initial young tree development that gives an earlier productivity
5. Soil temperature increase leading to a more efficient tree root development
6. Reduced environmental impact, as this technique completely substitutes chemical treatments
7. Good soil moisture keeping

compete with fruit crops, and their roots should be superficial to avoid interfering with those of fruit crops. Generally, to avoid or to limit these problems, spontaneous cover crops are replaced by artificial covering species, which develop quickly and with reduced water exigencies. Three to four species of gramineae or a mixture of gramineae and leguminoseae are often used in fruit tree crops to warrant a greater adaptation capability to environmental conditions.

Cover crops should be mowed when the grass is 10–15 cm high; the cut height should be 5–6 cm to favor sprouts; the time of intervention and cut height should be chosen in relation with species used for grass covering and with environmental conditions. Cover crop control can be carried out by mowing or shredding cover crops. During winter, groundcover vegetation should be harrowed at least once, and every three or four years, a complete scarification is necessary to hasten the oxidation of organic matter.

In fruit crops, it is necessary to implement grass covering when trees are completing their development, that is, after 2 or 3 years from orchard establishment.

Cover crops need nutritional element intake during the first years; then, grass mulching is generally enough to satisfy their mineral nutrition requirements.

CHEMICAL METHODS

Residual preemergence herbicides or non-residual postemergence herbicides, distributed as pure compounds or tank-mixed, are used to control weed development in fruit orchards.

Residual preemergence herbicides are sprayed on the ground prior to seed germination; they work by either blocking seed germination or killing germinating seedlings after root absorption. They remain in the soil's superficial layers and they slowly leach, showing a layer selectivity. The tendency to leach and the herbicide selectivity and efficiency depend on soil characteristics, soil water and organic matter contents, irrigation frequency, the herbicide method, time of application, etc. The most important preemergence herbicides are simazine, diuron, trifluralin, oxyfluorfen, and propyzamide.

Postemergence herbicides are absorbed by green aerial organs and they can either be transported or not in the plant. Non-transported herbicides destroy vegetative green parts and they are active on annual or biennial weeds; they do not have much definite effect on perennial weeds, which are temporarily devitalized but not definitively killed. Transported herbicides accumulate in annual or perennial weed reserve organs, inducing death. Non-transported herbicides that are often used include paraquat, diquat, and glyphosinate-ammonium; the transported herbicides

are MCPA, dicamba, dalapon, glyphosate, fluzifop-*p*-butyl, sulphosate, etc.

Fruit crop weed chemical control has been extensively developed around the 1950s with the use of residual herbicides (e.g., simazine). Later, postemergence herbicides (diquat and paraquat) and, around the 1970s, transported herbicides became widely used. Their wide activity spectrum made it possible to adapt weed control to many different cultural exigencies, which notably reduced the use of herbicides.

Thanks to the fruit crop orchard structure, i.e., the alternation of rows and free-vegetation areas, it has become possible to apply different weed control strategies. Chemical control under the rows and mechanical tillage in row-middle can adequately repress weed development; this strategy limits water and mineral nutrient competition and, by reducing the surface for herbicide treatment, reduces herbicide costs and quantities. In drought locations, ground tillage can improve soil water reserves. Some negative aspects tied to mechanical tillage, in particular the plow sole formation and Fe and B deficiency in chlorosis-sensitive species, can develop.

Weed chemical control under trees associated with cover crops within rows is diffused in cool climate regions among fruit crops that are not extremely sensitive to the antagonistic effects exerted by weeds. In the humid regions of North America and Europe, an arrangement of herbicides-treated tree rows with mowed-grass row-middle areas has become the most common groundcover management system. Cover crops between rows show advantages and disadvantages (see "cover Crops"); furthermore, during fruit crop establishment, the competition exerted by weeds can be too extreme, even if mineral intake is provided. This competitiveness implies reduced tree growth and delayed fruit production; in these circumstances, it is more useful to have cover crops within rows with mulch or to resort to chemical control. To reduce environmental impact, the tree-row groundcover vegetation can be managed with non-residual postemergence herbicides or regular mowing and not with residual herbicides.

A further reduction of herbicide use could be possible. By knowing the threshold at which the negative impacts of groundcover species outweigh their positive impacts on the soil/crop system, it could be possible to use chemical control only in the period when weeds can really cause damage to the tree crops.

BIOLOGICAL METHODS

It has been suggested that the utilization of plant pathogens for biological weed control be used and integrated into weed-management systems. Compatibility of microbial herbicides with chemical herbicides has been demonstrated, and it may be

possible to improve the efficacy of bioherbicides by applying chemicals at sublethal rates. Moreover, the use of herbicides in combination with biological agents may expand the spectrum of weed control in the field.

At present, only a few bioherbicides have been developed and authorized for agricultural use, and not all of them can be applied in tree crops.

BIBLIOGRAPHY

- Anonymous *Proceedings 3rd International Symposium on No-Tillage and Other Soil Management Techniques in Vines*; Montpellier, France, 1991; 458 pp.
- Boyetchko, S.M. Principles of biological weed control with microorganism. *HortScience* **1997**, *32*, 201–205.
- Bugg, R.L.; Waddington, C. Using cover crops to manage arthropod pests of orchards: A review. *Agric. Ecosyst. Environ.* **1994**, *50*, 11–28.
- Hipps, N.A.; Samuelson, T.J. Effects of long-term herbicide use, irrigation and nitrogen fertilizer on soil fertility in an apple orchard. *J. Sci. Food Agric.* **1991**, *55*, 377–387.
- Merwin, I.A.; Stiles, W.C. Orchard groundcover management impacts on apple tree growth and yield, and nutrient availability and uptake. *J. Am. Soc. Hortic. Sci.* **1994**, *119*, 209–215.
- Merwin, I.A.; Ray, J.A. Spatial and temporal factors in weed interference with newly planted apple trees. *HortScience* **1997**, *32*, 633–637.
- Welker, W.V.; Glenn, D.M. Sod proximity influences the growth and yield of young peach trees. *J. Am. Soc. Hortic. Sci.* **1989**, *114*, 856–859.

Fumigation

Abraham Gamliel

Department of Agricultural and Environmental Engineering, Laboratory of Pest Management Application, ARO Volcani Center, Bet Dagan, Israel

INTRODUCTION

Fumigation is a chemical approach to eliminate pests in soil before planting, in storage and for disinfection of commodities. Fumigation is defined as a method of applying volatile toxicants, which act at the vapor phase, and usually kills a wide spectrum of organisms, which are exposed to these agents. Fumigants have provided great benefits to agricultural production for many years. Worldwide, fumigant use is important for healthy crop production, shipment of pest-free products from countries, and for quarantine purposes. Fumigation of stored food and grains prevents contamination of the food with organisms and hazardous toxins and also contributes to human health. The benefits of fumigants to global food production and to human health resulted in an increase in the use of fumigation as common disinfection practice in the last decades. However, certain fumigants were found to possess negative traits, resulting in acute and chronic health hazards as well as environmental pollution. Therefore, the use of fumigants should be carefully examined, and methods and technologies for effective use with reduced environmental hazards should be developed.

FUMIGANTS AND THEIR TOXICITY

Crop health is a major factor in determining plant growth and production. The soilborne pests—nematodes, pathogenic bacteria and fungi, arthropods, and weeds—can cause severe damage to plants and can survive in soil for a long period. Certain pests infest stored products (seeds, grains, etc.) and can destroy them. Application of highly toxic fumigants is a common approach to killing soilborne pests before planting and ascertaining a healthy crop. Soil disinfection fumigants, such as methyl bromide, are widely used for soil fumigation in intensive agriculture and for commodity and postharvest quarantine treatments. They have a broad-spectrum activity against pests including fungi, bacteria, viruses, arthropods, nematodes, and weeds. The spectrum of pests, which is controlled by fumigants, is presented in Table 1. Having high vapor pressure fumigants penetrate commodities and soils to deep layers. They are characterized by short lethal

exposure periods and can be applied also at low temperatures. The aeration period to eliminate volatile residues before planting is short in most cases.

The toxic effect of a fumigant is a function of its concentration (C) and the exposure period (T). The mathematical product of concentration and time is a constant (k), which usually is referred to as the $C \times T$ product (CTP). It is well accepted that CTP, under certain limits, is the appropriate measure to express the relative toxicity of fumigants. CTP needed for effective control of a given pest is frequently dependent on the temperature and is reduced at increasing temperature. Implementation of the CTP concept is important when reduced dosages are applied for extended periods to achieve the same level of pest control. This enables, e.g., a reduction in dosage of methyl bromide by 50% by extending the exposure time, yet achieving a similar level of control.

Fumigants and the Environment

Fumigants are usually highly toxic, resulting in simultaneous control of a wide variety of pests. However, negative effects, i.e., eradication of beneficial organisms, and negative shifts in the biological equilibrium in the soil, are also possible. The increased environmental concern of these negative attributes became a major factor in triggering regulatory restrictions on the use of soil fumigants. In many countries, the use of fumigants, such as nematicides including 1,2-dibromochloropropane, ethylene dibromide, and 1,3-dichloropropene, has been discontinued. Furthermore, a worldwide phase-out of methyl bromide—the major soil fumigant—is currently underway, because it was listed by the Montreal protocol as a potential atmospheric ozone depletion substance. Few soil and structure fumigants are still available; however, their use and method of application should be carefully considered, if we want to still use them without negative impact on the environment.

Application Methods of Fumigants

Soil fumigants should be applied to well-prepared soil before planting. Most chemicals are injected into the

Table 1 Major fumigants and their use

| Fumigant | Chemical structure | Application | Target organisms |
|---------------------|--|--------------------------------------|--|
| 1,3-Dichloropropene | CH ₂ Cl-CH-CHCl | Soil | Free-living and root-knot nematodes, insects, weeds |
| Carbon bisulfide | CS ₂ | Soil | Fungi, insects, nematodes |
| Chloropicrin | CCl ₃ -NO ₂ | Soil | Fungi, free-living nematodes, insects, weeds |
| Dazomet | C ₅ H ₁₀ N ₂ S ₂ | Soil | Fungi, free-living nematodes, insects, weeds |
| Formaldehyde | HCOH | Soil, Structure | Viruses, bacteria, lower fungi, weeds |
| Metam sodium | CH ₃ NCS | Soil | Fungi, free-living nematodes, insects |
| Methyl bromide | CH ₃ -Br | Soil, quarantine, structure, storage | Fungi, free-living and root-knot nematodes, insects, weeds |
| Phosphine | PH ₃ | Storage | Insects, rodent pests |

soil in a liquid form to the desired depth with special application machinery. The chemical vaporizes in the soil and diffuses to reach every niche in which pests exist. Application of a fumigant through an irrigation system with delivery via water to deep soil layers is also common. Such application, however, requires special formulation of the fumigants. Moreover, special care should be taken in order to avoid penetration of the toxicants to the main water system. A fumigated field is usually covered with plastic mulch following fumigation to minimize gas escape. Standard polyethylene films are permeable to fumigants mostly, and the fumigants dissipate quickly by escaping through the film shortly after application. The use of impermeable films is an important factor in reducing fumigant emission into the atmosphere. Impermeable films used with an extended exposure time should result in the desired CTP with reduced dosage.

Fumigation and Integrated Pest Management

Combining fumigants with other control methods, can improve pathogen control, thereby enabling reduced dosage and emission as well as minimizing other negative environmental effects. Combined fumigants can result in an additive effect, when each method is directed to control a specific pathogen. For example, a combination of MB (which is weak bactericide) and formaldehyde can be used to control a complex of nematode and bacterial diseases. Combined treatment can also result in a synergistic effect, when one control agent increases the vulnerability of the pathogen to the other. Combination of fumigants at reduced dosage with non-chemical treatments, such as solar heating of soil (solarization), is also possible. Such combination can result in effective control of various diseases, which could not be controlled effectively by solarization or by the fumigant alone.

Integration of biological agents can also improve the fumigation effect. Application of antagonistic fungi and bacteria, such as *Trichoderma harzianum*, *Bacillus* spp., etc., (either by incorporating into soil or as seed coating) following fumigation results in their colonization of the soil before pathogen invasion and may so effectively control various soilborne diseases.

FUTURE CONCERN

The use of fumigants for healthy crop production relies on effective and safe use. This includes good preparation of the soil, good sanitation, effective and appropriate application technologies, and disease-free plant propagation. The limited number of fumigants, which are still available, emphasizes the need for effective application and combining methods for achieving this goal. In the near future, fumigants will still be needed, as viable alternatives are still unavailable. The use of technologies, such as gas-impermeable films, improved application; combining these with other methods of control, such as solarization and biocontrol agents, will minimize environmental hazards while supporting effective pest control.

[See also Biological Pest Controls; Fumigants; Optimizing Pesticide Application; Nematicides; Stored Food Pest Management; Microorganism Pests; Insect/Mite Pests and Plant Pathogens.]

BIBLIOGRAPHY

- Eshel, D.; Gamliel, A.; Katan, J.; Grinstein, A. Evaluation of soil fumigants on soilborne fungal pathogens in a controlled-environment system and in soil. *Crop Prot.* **1999**, *18*, 437–443.
- Gamliel, A.; Hadar, E.; Katan, J. Improvement of growth and yield of *Gypsophila paniculata* by solarization or fumigation of soil or container medium in continuous cropping systems. *Plant Dis.* **1993**, *77*, 933–938.

- Gamliel, A.; Grinstein, A.; Katan, J. Improved technologies to reduce emissions of methyl bromide from soil fumigation. In *Improved Application Technology for Reduction of Pesticide Dosage and Environmental Pollution*; Grinstein, A., Ascher, K.R.S., Mathews, G., Katan, J., Gamliel, A., Eds.; Phytoparasitica, 1997; Vol. 25, 21–30. Supplement.
- Gamliel, A.; Grinstein, A.; Peretz, Y.; Klein, L.; Nachmias, A.; Tsror, L.; Livescu, L.; Katan, J. Reduced dosage of methyl bromide for controlling verticillium wilt of potato in experimental and commercial plots. *Plant Dis.* **1997**, *81*, 469–474.
- Gamliel, A.; Grinstein, A.; Klein, L.; Cohen, Y.; Katan, J. Permeability of plastic films to methyl bromide: field study. *Crop Prot.* **1998**, *17*, 241–248.
- Klein, L. Methyl Bromide as Soil Fumigant. In *The Methyl Bromide Issue*; Bell, C.H., Price, N., Chakrabarti, B., Eds.; John Wiley & Sons: New York, 1996; 191–235.
- Munnecke, D.E.; Van Gundy, S.D. Movement of fumigant in soil, dosage response and differential effects. *Annu. Rev. Phytopathol.* **1979**, *17*, 405–429.
- Tomlin, C. *The Pesticide Manual*, 10th Ed.; Crop Protection Publication: Surrey, UK, 1994.

Genetics of Resistance and Plant Breeding

Guido Jach

Department George Coupland: Developmental Plant Biology, Max-Planck Institute for Plant Breeding Research, Cologne, Germany

INTRODUCTION

Crops that are resistant to pests play a major role in integrated pest management strategies. Their “built-in” protection abolishes the need of pesticide application and allows for environmentally sound agriculture. During the long history of breeding, numerous strategies that use methods such as mass and pure-line selection, hybridization, generation, and use of mutated plant varieties have been developed. Although breeding for resistance uses these same methodologies, it differs in that the detection of the desired phenotype requires performance of recurrent bioassays and field trials challenging the plants with a certain pest. Hence breeding for resistance to pests is even more time-consuming than breeding for other traits. This problem might be overcome by using genetic engineering. A first example of a successful realization of this approach is given by insect-resistant transgenic plants expressing the *Bacillus thuringiensis* toxin.

Although observation of differences in resistance among plant varieties date back to Theophrastus (372–287 BC), breeding for resistance began in earnest at the beginning of the 20th century. Knight (1799) was one of the first to identify resistant wheat varieties. Darwin (1868) found other examples among onions, grapes, and strawberries. The general acceptance of the parasitic nature of plant disease in the middle of the 19th century and the development of the scientific basis of plant breeding were the two initial events that established today’s resistance breeding.

BREEDING FOR RESISTANCE

Improving crop resistance by breeding integrates various disciplines such as botany, genetics, plant physiology, plant pathology, entomology, biochemistry, statistics, and computer science. This list includes design of suitable methods to evaluate breeding material (bioassays), identification of sources of resistance genes, generation of new strains combining these genes, and comparison of these new strains to present cultivars.

By aiming at maximized yield and quality, plant breeders often eliminate undesirable variation that detracts from this goal. As a consequence, the results

produce genetically uniform varieties. However, genetic uniformity of the crop vastly increases the probability of new pathogens and pests to evolve. For this reason, most varieties must usually be replaced within 3–5 years from the time of their widespread distribution. Nevertheless, cultivation of genetically uniform resistant varieties can be used if other means of plant disease control are possible or if the resistance is aimed against slow-moving soil pathogens that do not spread rapidly and widely enough to cause an epidemic. Slow spread allows time for the control of the disease by other means or the replacement of the variety with another one that is resistant to the new race.

Genetic resistance may be general or specific. General resistance (uniform resistance, non-specific resistance, horizontal resistance) is equally effective against all isolates of a pathogen or pest, whereas specific resistance (non-uniform resistance, vertical resistance) is effective only against certain races.

General resistance is caused by the joint action of many genes (polygenic resistance) and confers durable protection by involving combinations of defense mechanisms, which in sum are beyond the probable limits of pathogen or pest variability. Although universally present in wild and domesticated plants, general resistance is highest in wild plants and lowest in greatly “improved” varieties. In contrast, specific resistance is a result of the presence of one or few major resistance genes (mono- or oligogenic resistance) and may not be durable: Pests can quickly evolve new races; introduction of a new race-specific resistance gene can result in selection for the matching pest that is quickly becoming prevalent.

These disadvantages can be avoided in some crops by using multilines; that is, mixtures of individual varieties (lines or cultivars) that are agronomically similar but differ in their resistance genes, or varieties carrying multiple major resistance genes derived from those varieties (pyramiding).

Conventional breeding requires the presence of suitable resistance genes in sexually compatible plants. Thus other native or foreign commercial varieties, older varieties, wild plant relatives, and plant lines carrying induced mutations can serve as sources for resistance genes (gene pool). Individual plants that survive a severely diseased environment (survivor plants) are likely to possess a suitable resistance trait.

Useful genetic variation is not equally distributed around the globe. In some regions of the earth, the so-called centers of diversity (=centers of origin: the areas of greatest diversity), ancestral, or related forms of crop plants occur in abundance either in the wild or as primitive cultivars. It was theorized that the world's crops had originated in eight centers of origin: China; India (with a related center in Indo-Malaya); Central Asia; the Near East; the Mediterranean; Ethiopia; southern Mexico and Central America; and South America (Peru, Ecuador, Bolivia, Brazil, and Paraguay). The value of collecting and maintaining germ plasm from these centers is widely recognized. Regional and national plant introduction services maintain seed stocks and distribute clonally propagated materials to breeders.

RESISTANCE MECHANISMS

Plants protect themselves from insect pests using various strategies that can be described as non-preference, antibiosis, tolerance, and avoidance. Preformed defenses as well as inducible reactions are used to realize these strategies.

Non-preference mechanisms include variations of color, taste, shape, and fragrances of flowers that alter the apparency (the detection of plants by the pests) and make them undesirable for insects as food, location for reproduction, or shelter. Thus the plant develops morphological and chemical traits to alter capture, ingestion, and digestion of plant material by the pest, e.g., thorns, spines, trichomes, prickles, increased leaf toughness, and lowered nutritional value as a result of the synthesis of phenolics, silica, and other chemicals.

Antibiosis is based on the production of toxic metabolites affecting the insect's development and reproduction after eating the plant tissue.

Tolerance includes all mechanisms resulting in good performance of the plants even in the presence of the insect.

Avoidance is based on escaping by maturing before the epidemic develops.

According to current models, rapidly induced increases in secondary metabolites result from specific signals controlling the metabolic pathways that produce the chemical defenses. Jasmonates are candidate signal molecules that are able to induce major classes of secondary metabolites (phenolics, alkaloids, and terpenes) as well as numerous proteins involved in plant defense.

BREEDING STRATEGIES

Pure line or pedigree selection works via the selection and separate propagation of individual highly resistant

plants and their progenies involving repeated resistance tests. This method is easy and most effective with self-pollinated crops but quite difficult with cross-pollinated ones.

Mass selection of seed from the most highly resistant plants surviving in a field where natural infection regularly occurs represents another simple breeding method. However, plants slowly improve, and in cross-pollinated plants, there is no control of pollen source.

Hybridization is the most common procedure and works via crossing of a desirable, but susceptible, variety of a crop with another cultivated or wild relative that carries resistance to a particular pathogen. This is followed by the testing of the progeny for resistance. Controlled crossing includes numerous steps: prevention of self-pollination (emasculation: manual removal of anthers), exclusion of foreign pollen, collection of pollen, and pollination of the target plant. If available, breeders use varieties showing male sterility (pollen is absent or non-functional) or self-incompatibility (plants are unable to produce a zygote after self-pollination) to avoid tedious emasculating work. To stabilize the resistance in the genetic background of the desired variety, resistant individuals of the offspring are repeatedly back-crossed to the desirable variety. Hybridization is time-consuming, and its effectiveness considerably varies with each particular case. Its application is somewhat easier in cross-pollinated than in self-pollinated crops. However, strains of cross-pollinated species considerably differ in their performance in crosses (combining ability). This also has to be taken into account.

In mutation breeding, the above-mentioned methods are preceded by an additional step increasing the genetic variability of a given variety by artificially inducing mutants using UV light, X-rays, or chemicals.

Genetic engineering of plants (plant transformation) allows the transfer of genes from any foreign genetic source (DNA) into the genome of a target plant, thereby generating a transgenic plant. Clearly, this methodology overcomes some limitations of conventional breeding procedures: The gene pool for resistance genes is vastly enlarged; only one trait is transferred without changing others; the procedure is faster and requires less generations; and the method allows for novel traits. However, the use of genetic engineering in plants has its own drawbacks. If compliance issues (regulating the environment and the human consumption of genetically engineered foods) are considered, genetic engineering techniques may be more costly and labor-intensive than the conventional methods.

On the molecular level, a gene (transcription unit) represents a stretch of DNA consisting of a sequence that is transcribed to a functional RNA product and regulatory sequences controlling transcription.

The plant cell's protein biosynthesis apparatus uses the produced RNA to synthesize the encoded protein (translation). Proper gene expression requires transcription and translation to occur. Plant transformation involves isolation of a useful gene, transfer of the gene into plant cells, integration of the gene into the plant genome, regeneration of fertile plants, expression of the transgene in the regenerated plants, and transmission of the transgene from generation to generation.

Several methods have been developed to introduce genes into the chromosomal DNA of plant cells: direct DNA uptake, microinjection of DNA, lipid vesicle-mediated delivery of DNA, use of plant viral vectors, and, most importantly, use of the natural transfer system of *Agrobacterium tumefaciens*.

Transgenic plants expressing the delta endotoxin from *B. thuringiensis* (Bt-toxin) represent the first examples of genetically engineered crops possessing insect resistance. Clearly, for engineered insect resistance based on single genes, the limitations of specific (major gene) resistance mechanisms apply.

Besides genetic engineering, plant molecular biology offers molecular marker technology as a valuable tool

to allow plant breeders to perform marker-assisted selection (selection for marker genes that are known to be linked to the genes controlling the trait) and genetic fingerprinting. The latter may help breeders to distinguish cultivars and estimate the genetic relationship of plants (as reflected in DNA polymorphisms). Molecular markers include isozymes, DNA restriction fragment length polymorphisms (RFLPs), random amplified polymorphic DNA (RAPDs), amplified fragment length polymorphisms (AFLPs), simple sequence repeats (SSRs), or microsatellites.

BIBLIOGRAPHY

- Bent, A.F.; Yu, I.C. Applications of molecular biology to plant disease and insect resistance. In *Advances in Agronomy*; Sparks, D.L., Ed.; Academic Press, 1999; 251–298.
- Hayward, M.D., Bosemark, N.O., Romagosa, I., Eds.; *Plant Breeding: Principles and Prospects*; Chapman and Hall: London, 1993.
- Karban, R.; Baldwin, I.T. *Induced Responses to Herbivory*; The University of Chicago Press: Chicago, 1997.

Grape Production in Australia: Integrated Strategies and Bioindicators for Sustainability

Linda J. Thomson

Centre for Environmental Stress and Adaptation Research (CESAR), University of Melbourne, Parkville, Victoria, Australia

Ary A. Hoffman

Department of Zoology, Cooperative Research Centre for Viticulture (CRCV), Glen Osmond, South Australia, Australia

INTRODUCTION

In vineyards, invertebrate pests coexist with a wide range of natural enemies capable of exerting significant control through predation and parasitism. Best management practice for the goal of sustainable grape production seeks not only to minimize environmental effects on surrounding areas and recognize the role of agricultural landscapes in maintenance of biodiversity but also to sustain the natural enemies important to biological pest control. Increasing the richness or biodiversity of species assemblages is not only relevant for conservation but also has been claimed to increase their impact on pest control.^[1,2] The close relationship of invertebrates with the environment coupled with characteristics shared by many common invertebrates makes them a good choice for bioindication: Short life cycles lead to rapid response to changes in management practice,^[3] they constitute by far the largest portion of measurable biodiversity,^[4] and their measurement is cost effective in that simple low cost traps can be put in place to collect and assess numbers of many different taxa.

BENEFICIALS AND CHEMICALS IN AUSTRALIAN VINEYARDS

The interaction between pest species and control agents plays a major role in agricultural production, a role that is directly relevant to profitability. Naturally occurring invertebrates have helped control agricultural pest species resulting in billions of dollars saved annually worldwide.^[5] In vineyards in Australia, a large number of natural enemies contribute to control of pests such as light brown apple moth (LBAM) (*Epiphyas postvittana* Walker (Lepidoptera: Tortricidae), long-tailed mealybug (*Pseudococcus longispinus* (Targioni-Tozzetti) (Hemiptera: Pseudococcidae), grapevine scale *Parthenolecanium persicae* (Fabricius) (Hemiptera: Coccidae), and several mites: bunch

Brevipalpus sp. (Acarina: Tenuipalpidae), rust *Calepitrimerus vitis* (Nalepa) and blister *Colomerus vitis* (Pagenstecher) Acarina: Eriophyidae (Table 1).^[6,7]

In fact, often control problems only emerge when populations of natural enemies are suppressed or removed, either soon after application of a broad-spectrum insecticide or in the following season.^[7] Increases in populations of mealybugs,^[7] vine scale,^[11] and eriophyoid mites^[12] have all been associated with pesticide applications. Vineyard pests can be difficult to control with chemicals: eriophyoid mites spend most of the lifecycle protected inside leaf buds,^[13] mealybugs not only have a protective waxy coating but also hide under bark, adult female scales do not move and are hidden under a disc-like covering, and weevil larvae are entrenched in vine canes. Light brown apple moth larvae are frequently protected in webbed leaf rolls or grape bunches.^[14] In vineyards, sulfur, which is commonly used to control powdery mildew, reduces resident populations of parasitoid wasps such as *Trichogramma carverae* Oatman and Pinto, both by direct mortality and by reduction in fecundity.^[15] As the target host of *T. carverae* is light brown apple moth, a major vineyard pest, reduction in parasitoid numbers can negatively impact on pest suppression. Control failures of pests following applications of pesticides emphasise the need to both encourage an integrated approach to pest management and the use of sustainable management practices, which encourage control agents.

RESPONSES OF INVERTEBRATES TO MANAGEMENT PRACTICES IN VINEYARDS

It is not only pesticide use that impacts on invertebrates with the potential to contribute to natural control. The use of cultivation under vine and interrow, ground covers and cover crops, adjacent vegetation, canopy, and irrigation management all have the potential to extend their effects beyond that intended. We have examined responses of a wide range of

Table 1 Some pests and control agents recorded in Australian vineyards

| Pest | | Control agent | | |
|------------------------------|---|--------------------------------|---|-----------------------|
| Order: family | Name | Order: family | Name | Control method |
| Hemiptera: Coccidae | <i>Grapevine scale</i> ^[8] <i>P. persicae</i> | Hymenoptera: Encyrtidae | <i>Metaphycus</i> sp. | Parasitoid |
| | | Hymenoptera: Encyrtidae | <i>Crisatithorax</i> sp. | Parasitoid |
| | | Hymenoptera: Aphelinidae | <i>Coccophagus lymnia</i> | Parasitoid |
| | | Coleoptera: Coccinellidae | <i>Cryptolaemus montrouzieri</i> | Predator |
| | | Lepidoptera: Noctuidae | <i>Mataomena dubia</i> | Predator |
| Hemiptera: Pseudococcidae | Long tailed mealy bug ^[8] <i>P. longispinus</i> | Hymenoptera: Encyrtidae | <i>Anagyrus fusciventris</i> | Parasitoid |
| | | Hymenoptera: Encyrtidae | <i>Tetracnemoidea brevicornis</i> | Parasitoid |
| | | Hymenoptera: Encyrtidae | <i>T. sydneyensis</i> | Parasitoid |
| | | Hymenoptera: Encyrtidae | <i>Leptomastix</i> sp. | Parasitoid |
| | | Hymenoptera: Pteromalidae | <i>Ophelosia</i> sp. | Parasitoid |
| | | Hymenoptera: Aphelinidae | <i>Coccophagus gurneyi</i> | Parasitoid |
| | | Coleoptera: Coccinellidae | <i>Rhizobius ruficollis</i> | Predator |
| | | Coleoptera: Coccinellidae | <i>Cryptolaemus montrouzieri</i> | Predator |
| | | Coleoptera: Coccinellidae | <i>Callineda testudinaria</i> | Predator |
| | | Neuroptera: Chrysopidae | <i>Pleiochrysa ramburi</i> | Predator |
| | | Neuroptera: Chrysopidae | <i>Chrysops</i> sp. | Predator |
| | | Neuroptera: Chrysopidae | <i>Chrysoperlae</i> sp. | Predator |
| | | Diptera: Cecidomyiidae | <i>Diadiplosis koebelei</i> | Predator |
| Hemiptera: Lygaeidae | Rutherglen bug (native) <i>Nysius vinitor</i> | Hymenoptera: Trichogrammatidae | <i>T. funiculatum</i> <i>T. carverae</i> | Egg parasitoid |
| Lepidoptera: Tortricidae | LBAM (native) <i>E. postvittana</i> Walker | Hymenoptera: Braconidae | <i>Ascogaster</i> sp. | Egg-larval parasitoid |
| | | Hymenoptera: Braconidae | <i>Phanerotoma</i> sp. | Egg-larval parasitoid |
| | | Hymenoptera: Braconidae | <i>Apanteles</i> sp. <i>A. tasmanica</i> | Larval parasitoid |
| | | Hymenoptera: Braconidae | <i>Bracon</i> sp. | Larval parasitoid |
| | | Hymenoptera: Braconidae | <i>Dolichogenidea tasmanica</i> | Larval parasitoid |

(Continued)

Table 1 Some pests and control agents recorded in Australian vineyards (Continued)

| Pest | | Control agent | | |
|------------------------|--|----------------------------------|---|-------------------------------------|
| Order: family | Name | Order: family | Name | Control method |
| | | Hymenoptera: Chalcidae | <i>Brachymeria phya</i> | Pupal parasitoid |
| | | Diptera: Tachinidae | <i>Voriela uniseta</i> | Larval parasitoid |
| | | Spiders (Araneae) ^[9] | | Predators |
| | | Hemiptera: Pentatomidae | Predatory shield bug | Larval predator |
| | | | <i>Oechalia schellenberg</i> | |
| | | Neuroptera: Hemerobiidae | <i>Micromus</i> sp. | Predator—eggs young caterpillars |
| | | | | Predator |
| Lepidoptera: Noctuidae | Grapevine moth <i>Phalaenoides glycinae</i> | Hemiptera: Pentatomidae | <i>Oechalia schellenbergii</i> | Predator |
| | | Hymenoptera: Eulophidae | <i>Euplectrus agaristae</i> | Larval parasitoid |
| | | Hymenoptera: Ichneumonidae | <i>Echthromorpha intricatoria</i> | Pupal parasitoid |
| | | Hymenoptera: Ichneumonidae | <i>Lissopimpla semipunctata</i> | Pupal parasitoid |
| Acarina: | Bunch mite <i>Brevipalpus</i> sp. | Acarina: Phytoseiidae | <i>Typhlodromus doreenae</i> | Predator |
| Tenuipalpidae | | | | |
| | | Acarina: Phytoseiidae | <i>Euseius victoriensis</i> (<i>Amblyseius victoriensis</i>) | Predator |
| Acarina: | Rust mite <i>C. vitis</i> | Acarina: Phytoseiidae | <i>Typhlodromus doreenae</i> | Predator |
| Eriophyidae | | | | |
| | | Acarina: Phytoseiidae | <i>Euseius victoriensis</i> | Predator |
| Acarina: | Blistar mite <i>C. vitis</i> | Acarina: Phytoseiidae | <i>Typhlodromus doreenae</i> | Predator |
| Eriophyidae | | | | |
| | | Acarina: Phytoseiidae | <i>Euseius victoriensis</i> | Predator |
| | | Acarina: Phytoseiidae | <i>A. loxtoni</i> | Predator |

| | | | | |
|------------------------------|--|--|--|---------------------------|
| Acarina: Tetranychidae | Two spotted mite <i>Tetranychus urticae</i> | Neuroptera: Hemerobiidae | Brown and green lacewings | Predator |
| Lepidoptera: Noctuidae | Pink cutworm <i>Agrotis munda</i> | Neuroptera: Chrysopidae Acarina: Phytoseiidae | <i>Galendromus occidentalis</i> | Predator |
| Coleoptera: Curculionidae | Introduced Apple weevil <i>Otiorhynchus cribricollis</i> | Coleoptera: Coccinellidae | <i>Stethorus</i> sp. | Predator |
| Coleoptera: Curculionidae | Introduced Garden weevil <i>Phytocinus callosus</i> | Dermaptera: Labiduridae ^[10] | <i>Labidura truncata</i> | Predator |
| Coleoptera: Curculionidae | Native vine weevil <i>Orthorhinus klugi</i> | Coleoptera: Carabidae | | Predator |
| Coleoptera: Curculionidae | Native elephant weevil <i>Orthorhynchus cylindrirostris</i> | Coleoptera: Staphylinidae | | Predator |
| Coleoptera: Curculionidae | Fruit tree root weevil <i>Leptoptis squalidus</i> | Araneae | | Predator |
| Coleoptera: Cerambycidae | Fig longicorn <i>Acalolepta vastator</i> | Neuroptera: Chrysopidae | Green lacewings | Predator |
| Thysanoptera | Thrips | Neuroptera: Hemerobiidae | Brown lacewings <i>Hemerobius</i> sp. | Predator |
| Orthoptera | Grasshoppers, locusts Katydid | Hemiptera: Nabidae | <i>Nabis</i> sp. | Predator |
| Dermaptera: Forficulidae | European earwig <i>Forficula auricularia</i> | Hemiptera: Reduviidae | | Predator |
| Sigmurethra | Snails | Diptera: Cecidomyiidae | <i>Diadiplosis koebelei</i> | Predator |
| | | Diptera: Syrphidae | Hoverflies | Predators and parasitoids |
| | | Diptera: Tachinidae | | Parasite |

(From Refs.^[6-10,29-31])

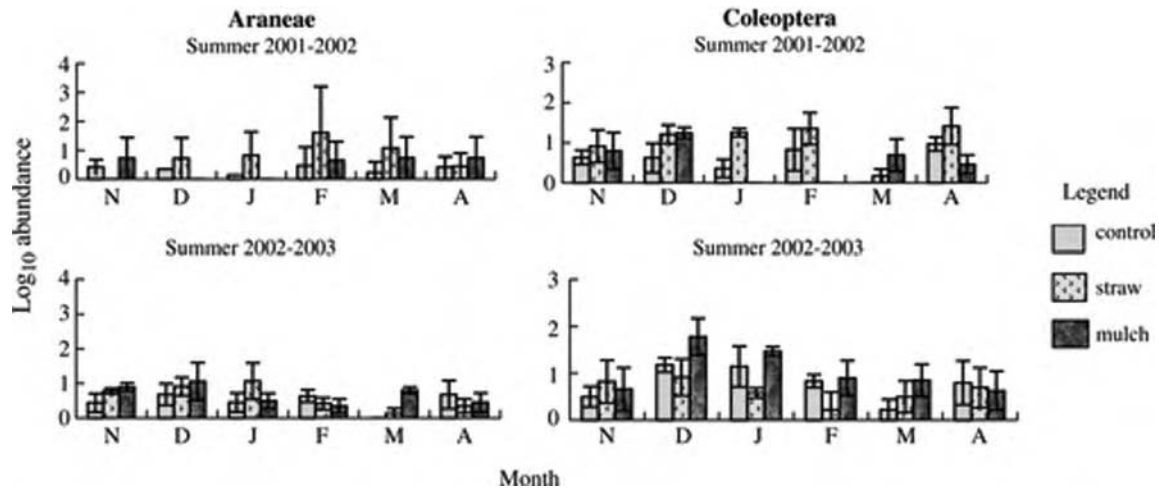


Fig. 1 Differences in response of Araneae and Coleoptera caught on the ground (pitfall traps) to management practices in seasons (winter compared to summer) and in summers with different climates in a vineyard trialing different ground cover treatments (straw, mulch, and bare earth). Five replicates for each treatment, sampling points 7 m apart. Error bars represent standard error.

invertebrates to different management practices in Southeastern Australian vineyards. Our initial studies have collected a wide range of invertebrates, and the results here discuss effects seen at the order level. Here we discuss effects on orders, which potentially contain predators and parasitoids. Our studies typically use ground level (pitfall) and canopy (sticky) traps.^[16]

Results in our trial of ground cover (mulch and straw compared to bare earth) where spiders and beetles were collected more frequently with straw (Fig. 1) were consistent with other studies: Ground cover such as mulch or straw has been found to increase density of spiders^[17] and beetles^[18] in other agroecosystems. Soil moisture is an important factor in determining the suitability of a site for ground dwelling beetles and spiders^[19,20] and our results show reduced irrigation

affected numbers of spiders and beetles collected on the ground but not in the canopy (Fig. 2).^[21] Beetle and spider numbers were both lower with practices that reduce irrigation.

In Australia, vineyards are often adjacent to vegetation as a consequence of recent clearing or deliberate conservation efforts. Field margins play an important agricultural role in providing a refuge for beneficial invertebrates (predators and parasitoids),^[22,23] and adjacent vegetation influences the numbers of invertebrates in vineyards. In a recent survey on remnant vegetation in a vineyard in the Yarra Valley, we found that there were higher numbers of beetles, predatory mites, and spiders in the more diverse vegetation adjacent to the vineyard, and that these were associated with higher numbers in the adjacent vineyard, extending to varying

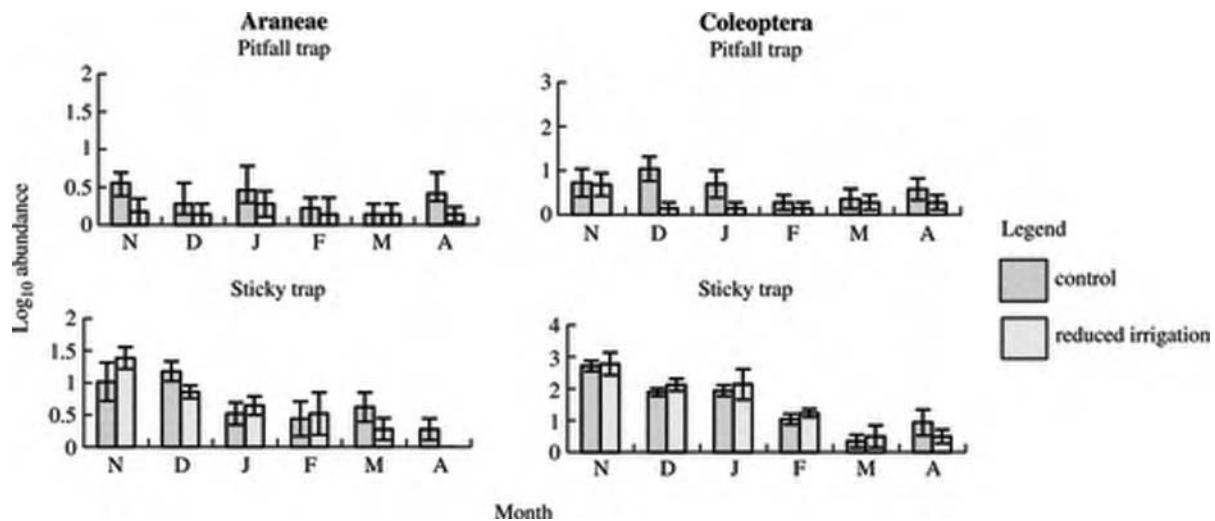


Fig. 2 Reduced numbers of spiders and beetles (pest and beneficial) caught at ground level (pitfall traps) but not in the canopy. Error bars are standard errors.

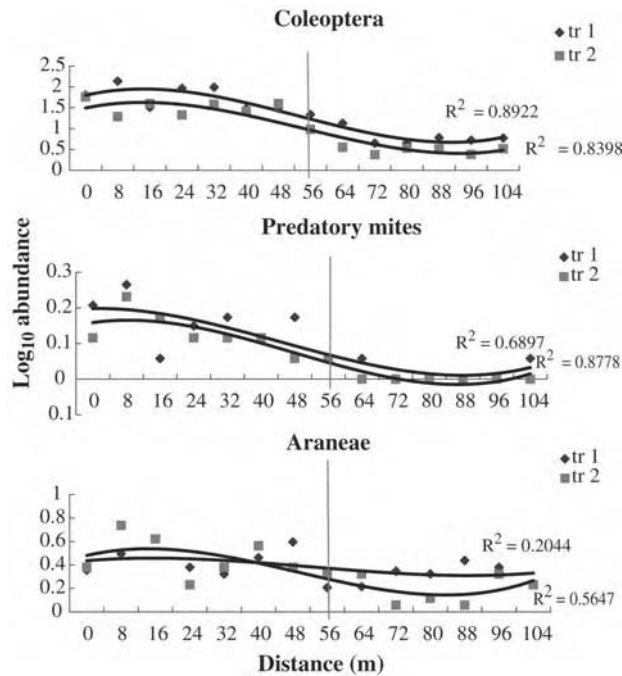


Fig. 3 Abundance of insects (log transformed average per trap) at points 7 m apart in two transects (tr 1, tr 2) extending from remnant vegetation (left hand side) into vineyards (right hand side). The boundary between the remnant and the vegetation is at 56 m (line). Lines represent non-linear regression incorporating cubic terms.

distances into the vineyard (Fig. 3). Choice of management practice can therefore have broad effects on numbers of organisms with potential to contribute to natural pest control in vineyards.

An alternative measure of effect of management practice on predator and parasitoid numbers is to measure activity. We have placed light brown apple moth egg masses at our replicated treatment sites to assess rates of predation and parasitism and found increased rates with ground cover and adjacent vegetation.

ORDER LEVEL OR LOWER LEVEL INDICATORS?

An important issue in biomonitoring is whether to sort to a fine scale level or sort at the coarse level of family/order.^[24,25] Sorting to order or family level for some orders can be relatively rapid but even to family level can be prohibitively time consuming. Collecting and sorting effort may require a choice to be made between assessing the impact on a wide range of organisms and a smaller number chosen for some predetermined reason such as their role as predators (e.g., carabids,^[26] soil conditioning (e.g., collembolan)^[27] or ubiquity (e.g., ants).^[28] In reality, systems will always be more complex—for example, many predators will contribute

to pest control. Sorting at order level allows sorting of a large number of organisms with a range of roles. Assessment on a narrow range of organisms has limited representative value and the question becomes one of identifying organisms to the level of taxonomic resolution necessary to satisfy the objectives of the study. How useful is order level information on the likely impact on pest control in vineyards? Identifying practices, which increase some orders may be adequate or at least informative, for example, identifying effects that increase numbers of Neuroptera, Dermaptera, Araneae, Opilionidae, and parasitoid Hymenoptera, because they are predators or parasitoids with the potential to contribute to pest control in vineyards and other agricultural ecosystems. However, for others, the value of order level information is questionable: While Coleoptera includes important families Carabidae, Staphylinidae, and Coccinellidae, it also includes weevils (Curculionidae), a local but occasionally significant vineyard pest. Hemiptera likewise includes important predatory families but also vinescale and mealybugs (Hemiptera: Coccidae and Pseudococcidae), and only a few dipteran families contribute to pest control in vineyards although some of these can be quite important (Cecidomyiidae, Asilidae, Syrphidae, and Tachinidae).

Hence while sorting large collections to order may give an indication of possible effects of management practices on pest control, sorting to at least family is essential for any measure of likely influence.

CONCLUSIONS

Our studies show that orders do respond differently to non-chemical management practices in vineyards and suggest groups of invertebrates susceptible to these practices. Identification of farm management practices, which enhance populations of natural enemies will increase natural control, help reduce reliance on chemical controls and increase sustainability of agricultural production. The result will be a reduction of inputs necessary without loss of production and negative impacts on viability of enterprise. The challenge is to show directly that the conservation of potential beneficials allows reduction in chemical use while maintaining production via control of pests.

ACKNOWLEDGMENTS

This research was supported by the Commonwealth Cooperative Research Centre (CRC) Program and conducted through the CRC for Viticulture with support from Australia's grapegrowers and winemakers through their investment body the Grape and Wine Research and Development Corporation with

matching funds from the federal government. Infrastructure support for this research was provided by the Centre for Environmental Stress and Adaptation Research funded by the Australian Research Council.

REFERENCES

- Altieri, M.A. The ecological role of biodiversity in agroecosystems. In *Invertebrate Biodiversity as Bioindicators of Sustainable Landscapes. Practical Uses of Invertebrates to Assess Sustainable Land Use*; Paoletti, M.G., Ed.; Elsevier: Amsterdam, 1999; 19–32.
- Cardinale, B.J.; Harvey, C.T.; Gross, K. Biodiversity and biocontrol: emergent impacts of a multi-enemy assemblage on pest suppression and crop yield in an agroecosystem. *Ecol. Lett.* **2003**, *6*, 857–865.
- Golden, D.M.; Crist, T.O. Experimental effects of habitat fragmentation on old-field canopy insects: community, guild and species responses. *Oecologia* **1999**, *118*, 371–380.
- Duelli, P.; Obrist, M.K. Biodiversity indicators: the choice of values and measures. *Agric. Ecosyst. Environ.* **2003**, *98*, 87–98.
- Pimentel, D., Ed.; *Techniques for Reducing Pesticide Use: Economic and Environmental Benefits*; Wiley: New York, 1997.
- Buchanan, G.A.; Amos, T.G. Grape pests. In *Viticulture. Volume 2 Practices*; Coombe, B.G., Dry, P.R., Eds.; Winetitles: Adelaide, 1992; 209–231.
- Nicholas, P., Margarey, P., Watchel, M., Eds.; *Diseases and Pests. Grape Production Series Number 1*; Winetitles: Adelaide, 1998.
- Waterhouse, D.F.; Sands, D.P.A. *Classical Biological Control of Arthropods in Australia*; CSIRO Entomology: Canberra, 2001.
- Danthanararayana, W. Population ecology of the light brown apple moth, *Epiphyas postvittana* (Lepidoptera: Tortricidae). *J. An. Ecol.* **1983**, *52*, 1–33.
- Horne, P.A.; Edward, C.L. Phenology and food preferences of *Labidura truncata* Kirby (Dermaptera: Labiduridae) in western Victoria. *J. Aust. Entomol. Soc.* **1995**, *34*, 101–104.
- Grafton-Cardwell, E.; Ping, G. Conserving vedalia beetle, *Rodolia cardinalis* (Mulsant) (Coleoptera: Coccinellidae), in citrus: a continuing challenge as new insecticides gain registration. *J. Econ. Entomol.* **2003**, *96*, 1388–1398.
- Whitney, J.; James, D.G. Doreen and Victoria: viticultural supermites. In *Pest Control and Sustainable Agriculture*; Corey, S.A., Dall, D.J., Milne, W.M., Eds.; CSIRO: Melbourne, Canberra, 1992; 240–241.
- Bernard, M.B.; Horne, P.; Hoffmann, A.A. Preventing restricted spring growth (RSG) in grapevines by successful rust mite control: spray application, timing and eliminating sprays harmful to rust mite predators are critical. *Aust. Grapegrow. Winemak.* **2001**, *453*, 26.
- Thomson, L.J.; Bennett, D.M.; Glenn, D.C.; Hoffmann, A.A. Developing *Trichogramma* as a pest management tool. In *Predators and Parasitoids*; Koul, O., Dhaliwal, G.S., Eds.; Taylor and Francis: London, 2003; 65–85.
- Thomson, L.J.; Glenn, D.C.; Hoffmann, A.A. The effects of sulfur on *Trichogramma* egg parasitoids in vineyards: measuring toxic effects and establishing release windows. *Aust. J. Exp. Agric.* **2000**, *40*, 1165–1171.
- Thomson, L.J.; Neville, P.J.; Hoffmann, A.A. Effective trapping methods for assessing invertebrates in vineyards. *Aust. J. Exp. Agric.* **2004**, *44*, 947–953.
- Nyffeler, M.; Sunderland, K.D. Composition, abundance and pest control potential of spider communities in agroecosystems: a comparison of European and US studies. *Agric. Ecosyst. Environ.* **2003**, *95*, 579–612.
- Holland, J.M.; Luff, M.L. The effects of agricultural practices on Carabidae in temperate agroecosystems. *Integ. Pest Man. Rev.* **2000**, *5*, 109–129.
- Frampton, G.K.; van den Brink, P.J.; Gould, P.J.L. Effects of spring drought and irrigation on farmland arthropods in southern Britain. *J. App. Ecol.* **2000**, *37*, 865–883.
- Perner, J.; Malt, S. Assessment of changing agricultural land use: response of vegetation, ground-dwelling spiders and beetles to the conversion of arable land into grassland. *Agric. Ecosyst. Environ.* **2003**, *98*, 169–181.
- Thomson, L.J. Influence of reduced irrigation on beneficial invertebrates in vineyards. *Aust. J. Exp. Agric.* **2006**, *46* (10), *in press*.
- Szentkiralyi, F.; Kozar, F. How many species are there in apple insect communities? Testing the resource diversity and intermediate disturbance hypotheses. *Ecol. Entomol.* **1991**, *16*, 491–503.
- Landis, D.A.; Wratten, S.D.; Gurr, G.M. Habitat management to conserve natural enemies of arthropod pests in agriculture. *Ann. Rev. Entomol.* **2000**, *45*, 175–201.
- Pik, A.J.; Oliver, I.; Beattie, A.J. Taxonomic sufficiency in ecological studies of terrestrial invertebrates. *Aust. J. Ecol.* **1999**, *24*, 555–562.
- Hilty, J.; Merenlender, A. Faunal indicator taxa selection for monitoring ecosystem health. *Biol. Conserv.* **2000**, *92*, 185–197.
- Kromp, B. Carabid beetles in sustainable agriculture: a review on pest control efficacy, cultivation impacts and enhancement. In *Invertebrate Biodiversity As Bioindicators of Sustainable Landscapes. Practical Uses of Invertebrates to Assess Sustainable Land Use*; Paoletti, M.G., Ed.; Elsevier: Amsterdam, 1999; 19–32.
- Ponge, J.F.; Gillet, S.; Dubs, F.; Fedoroff, E.; Haese, L.; Sousa, J.P.; Lavelle, P. Collembolan communities as bioindicators of landuse intensification. *Soil Biol. Biochem.* **2003**, *35*, 813–826.
- Anderson, A.N.; Hoffmann, B.D.; Muller, W.J.; Griffiths, A.D. Using ants as bioindicators in land management: simplifying assessment of ant community responses. *J. App. Ecol.* **2002**, *39*, 8–17.
- CSIRO. *Insects of Australia*. Melbourne University Press: Melbourne, 1991.
- Emmett, R.W.; Harris, A.R.; Taylor, R.H.; McGechan, J.K. Grape diseases and vineyard protection. In *Viticulture. Volume 2 Practices*; Coombe, B.G., Dry, P.R., Eds.; Winetitles: Adelaide, 1992; 232–278.
- Geest, L.P.S., Evenhuis, H.H., Eds.; *Tortricid Pests and their Biology, Natural Enemies and Control. World Crop Pests*; Elsevier: Amsterdam, 1991.

Grapes and Insects: Ecology and Control

Walter J. Bentley

Kearney Agricultural Center, University of California, Statewide IPM Program,
Parlier, California, U.S.A.

Lucia Varela

University of California Cooperative Extension, Sonoma County,
Santa Rosa, California, U.S.A.

Kent M. Daane

Department of Environmental Science, Policy, and Management, Kearney Agricultural Center,
University of California-Berkeley, Berkeley, California, U.S.A.

INTRODUCTION

The grape is one of the oldest cultivated crops in the world. Grape culture originated in Asia Minor, between the Black and Caspian seas in about 6000 B.C.^[1] This area is the home of *Vitis vinifera*,^[2] the predominant commercial cultivar. From there, grapes were spread throughout Europe. Although North America has native grape species (*Vitis girdiana*, Munson; *Vitis labrusca*, Linnaeus; *Vitis rupestris*, Scheele; and others), *V. vinifera* was brought to the New World with Spanish explorers and was first grown in California at Mission San Francisco Xavier in 1697.^[3] Grapes are grown for fresh consumption, dried as raisins, or crushed for wine and juice production. Worldwide, there is an estimated 19,000,000 acres planted.^[4]

ARTHROPOD PESTS OF GRAPE

Approximately 150 species of arthropods are considered pests of grape worldwide.^[2] In the United States, there are about 60 arthropod pests. The most severe (phylloxera, vine mealybug, and leafhoppers) are known worldwide. Table 1 presents a selected list of recorded arthropod pests of grape.

ECONOMIC IMPACT OF GRAPE PESTS

The presence and relative importance of a pest varies from one area to another, depending on crop market and environmental conditions. For example, moderate infestations of grape mealybug, *Pseudococcus maritimus* (Ehrhorn), would not be a problem in wine grapes but would make table grapes unmarketable. Similarly, Pacific mite, *Tetranychus pacificus* McGreggor, is considered a major pest of grape in central California

where defoliation is possible due to feeding, but is occasionally a problem along the north coast of California, Washington, and Oregon. Consequently, integrated pest management (IPM) programs may vary for the same pest.

Few arthropod grape pests kill vines. Exceptions to this include grape phylloxera (*Daktulosphaira vitifoliae*) and glassy-winged sharpshooter (*Homalodisca coagulata*), vector of the bacteria (*Xyllela fastidiosa*), causal agent of Pierce's disease. *X. fastidiosa* has killed whole vineyards in southern California and in the southeastern United States. Some pests, such as the Achemon sphinx moth, *Eumorpha achemon* (Drury), can completely defoliate a vineyard and thereby destroy a single year's production. Similarly, vine mealybug can result in complete infestation of clusters, making the crop unmarketable.^[5,6]

Pesticide treatment costs for pests such as variegated leafhopper, vine mealybug, omnivorous leafroller, and Pacific and Willamette spider mites range from \$25 to \$150 per acre in the United States.

CASE STUDY GRAPE PEST MANAGEMENT IN CALIFORNIA

Integrated pest management of insects and mites has progressed greatly during the last 30 years, particularly in California. Knowledge of pest and beneficial arthropods in the system, good sampling methods (spider mites, leafhoppers, mealybugs, and cutworms) and accurate treatment thresholds (spider mites, leafhoppers, and mealybugs) have been developed for key pests in California.^[5] Integrated pest management in California grape production utilizes biological, cultural, mechanical and physical, chemical and regulatory control strategies to manage arthropod pests.

Table 1 Grape insects of the world

| Plant organ attacked | Pest | | |
|--------------------------------|--|---------------------------------|-----------------------------|
| | Order | Region | |
| Roots | Hemiptera | | |
| | <i>Daktulosphaira (Viteus) vitifoliae</i> | World | |
| | <i>Cicada</i> spp. | South Europe | |
| | <i>Tibicen haematodes</i> | South Europe | |
| | <i>Magicicada septemdecim</i> | California | |
| | <i>Rhizoecus falcifer</i> | California | |
| | <i>Margarodes meridionalis</i> | California | |
| | <i>Margarodes vitis</i> | Chile | |
| | <i>Margarodes capensis</i> | South Africa | |
| | <i>Margarodes greeni</i> | South Africa | |
| | <i>Eurhizoecus brasiliensis</i> | Brazil | |
| | Coleoptera | | |
| | <i>Bromius obscurus</i> | Europe, North America | |
| | <i>Fidia viticida</i> | North America | |
| | <i>Scelodonta strigicollis</i> | India | |
| | <i>Vesperus</i> spp. | France, Spain, Italy | |
| | <i>Pentodon</i> spp. | South Europe | |
| | <i>Phyllognatus excavatus</i> | South Europe | |
| | <i>Opatrum sabulosum</i> | Europe | |
| | Lepidoptera | | |
| | <i>Vitacea polistiiformis</i> | Missouri | |
| | Wood trunk | Isoptera | |
| | | <i>Calotermes flavicollis</i> | South Europe |
| | | <i>Reticulitermes lucifugus</i> | South Europe |
| | | <i>Reticulitermes hesperus</i> | California |
| | | <i>Incisitermes minor</i> | California |
| | | Lepidoptera | |
| <i>Cossus cossus</i> | | South Europe | |
| <i>Paropta paradoxus</i> | | Israel, Egypt | |
| Coleoptera | | | |
| <i>Anaglypytus mysticus</i> | | Bulgaria | |
| Shoots | | Hemiptera | |
| | | <i>Parthenolecanium corni</i> | United States, South Europe |
| | <i>Pulvinaria innumerabilis</i> | California | |
| | <i>Pulvinaria vitis</i> | South Europe | |
| | <i>Diaspidiotus wvae</i> | California | |
| | <i>Homolodisca coagulata</i> | United States | |
| | <i>Ceresa bubalus</i> (Membracidae) | Eastern United States | |
| | Coleoptera | | |
| | <i>Apate tenebreans</i> | Africa | |
| | <i>Stenias grisator</i> | India | |
| | <i>Synoylonb anale</i> | South Europe, Asia | |
| | Hymenoptera | | |
| | <i>Macrophya strigosa</i> (Tenthredinidae) | South Europe | |
| | Coleoptera | | |
| | <i>Polycaon confertus</i> (Coleoptera) | Western United States | |
| | Buds and very young shoots | Lepidoptera | |
| | | <i>Arctia caja</i> | South Europe |
| | | Noctuidae | World |
| Coleoptera | | | |
| <i>Peritelus sphaeroides</i> | | South Europe | |
| <i>Peritelus noxius</i> | | South Europe | |
| <i>Glyptoscelis squamulata</i> | California | | |
| <i>Paracotalpa ursina</i> | California | | |
| <i>Phlyctinus callosus</i> | South Africa | | |

(Continued)

Table 1 Grape insects of the world (Continued)

| Plant organ attacked | Pest | | |
|------------------------------------|---|------------------------------|--|
| | Order | Region | |
| Leaf | <i>Eremnus cerealis</i> | South Africa | |
| | <i>Eremnus stulosus</i> | South Africa | |
| | <i>Limoniuss canus</i> | Western United States | |
| | <i>Paracotalpa ursina</i> | California | |
| | <i>Altica torquata</i> | California | |
| | Acarina | | |
| | <i>Calepitrimerus vitis</i> | South Europe, USA | |
| | <i>Eriophyes oculivitis</i> | Egypt | |
| | <i>Eriophyes vitineusgemma</i> | Moldavia | |
| | <i>Brevipalpus lewisi</i> | California | |
| | Lepidoptera | | |
| | <i>Desmia funeralis</i> | North America | |
| | <i>Sparganothis pilleriana</i> | South Europe | |
| | <i>Hyles lineate</i> | Europe, North America | |
| | <i>Eumorpha achemon</i> | California | |
| | <i>Antispila rivillei</i> | Georgia, Turkey, Iran | |
| | <i>Harrisian brilliams</i> | Mexico, California | |
| | <i>Sylepta ovalis</i> | India, Kenya | |
| | <i>Etigmene acraea</i> | California | |
| | <i>Spodoptera exigua</i> | California | |
| | <i>Spodoptera praefica</i> | California | |
| | <i>Therta alecto</i> | Egypt, Iran, Lebanon | |
| | <i>Therisimima ampelophaga</i> | Europe | |
| | Coleoptera | | |
| | <i>Haltica lythri</i> subsp. <i>ampelophaga</i> | Europe | |
| | <i>Haltica chalybea</i> | United States | |
| | <i>Haltica torquata</i> | California | |
| | <i>Haltica ampelophaga</i> | Greece | |
| | <i>Byctiscus betulae</i> | Europe | |
| | <i>Hoplia callipyge</i> | California | |
| | <i>Otiorhynchus sulcatus</i> | California | |
| | <i>Adoretus punctipenis</i> | India | |
| | <i>Epaticier arachniiformis</i> | Lebanon | |
| | <i>Phycinus callosus</i> | South Africa | |
| | <i>Scelodnota strigicollis</i> | India | |
| | Dermaptera | | |
| | <i>Forficula auricularia</i> | California | |
| | Hemiptera | | |
| | <i>Aleurocanthus spiniferus</i> | Asia, Africa, Hawaii | |
| | <i>Aphis illinoisensis</i> | Eastern United States | |
| | <i>Aphis citricoli</i> | California | |
| | <i>Aphis gossipyii</i> | California | |
| | <i>Philaenus spumarius</i> | North America, Europe | |
| | <i>Scaphoideus littoralis</i> | North America, France, | |
| | | Germany, Switzerland, Italy | |
| | <i>Empoasca flavescens</i> | Europe | |
| | <i>Empoasca lybica</i> | Spain, South Italy, Maghreb, | |
| | Tanganyka | | |
| <i>Flata ferrugata</i> | Punjab | | |
| <i>Umnata intracta</i> | Punjab | | |
| <i>Zygina rhamni</i> | France | | |
| <i>Erythronera adanae vitisuga</i> | Bulgaria | | |
| <i>Erythronera comes</i> | California | | |
| <i>Erythronera variabilis</i> | California | | |
| <i>Erythronera elegantulae</i> | California | | |
| <i>Erythronera ziczac</i> | British Columbia | | |
| <i>Draeculacephala minerva</i> | California | | |
| <i>Graphocephala atropumctata</i> | California | | |
| <i>Carnecephala fulgida</i> | California | | |

(Continued)

Table 1 Grape insects of the world (Continued)

| Plant organ attacked | Pest | |
|---|---|-----------------|
| | Order | Region |
| Gall makers | Heteroptera | |
| | <i>Nyzius senecionis</i> | France |
| | <i>Nysius raphanus</i> | Europe, America |
| | Acarina | |
| | <i>Eotetranychus carphini</i> | South Europe |
| | <i>Panonychus ulmi</i> | South Europe |
| | <i>Brevipalpus lewisi</i> | Bulgaria, USA |
| | <i>Tetranychus pacificus</i> | California |
| | <i>Eotetranychus willamettei</i> | California |
| | <i>Tetranychus atlanticus</i> | France |
| | <i>Oligonychus mangiferae</i> | India |
| | Acarina | |
| | <i>Eriophyes vitis</i> | South Europe |
| | <i>Eriophyes vitigenusgemma</i> | Moldavia |
| | <i>Eriophyes oculivitis</i> | Egypt |
| Hemiptera | | |
| <i>Daktylosphaira (viteus) vitifoliae</i> | World | |
| Diptera | | |
| <i>Janetiella oenophila</i> | France, Italy | |
| <i>Lasioptera vitis</i> | Eastern United States | |
| <i>Dasyneura vitis</i> | Eastern United States | |
| <i>Schizomyia pomorum</i> | Eastern United States | |
| Fruits | | |
| Lepidoptera | | |
| <i>Clysiana ambiguella</i> | Europe, Asia, Russia, Brazil, Turkey | |
| <i>Lobesia botrana</i> | Europe | |
| <i>Lobesia viteana</i> | Eastern United States | |
| <i>Argyrotaenia citrana</i> | California | |
| <i>Argyrotaenia politana</i> | France | |
| <i>Argyrotaenia velutinana</i> | Eastern United States | |
| <i>Platynota stultana</i> | California | |
| <i>Phalaenoides glycine</i> | Australia | |
| <i>Epiphyas postvittana</i> | Australia | |
| <i>Serodes partitus</i> | South Africa | |
| <i>Crytoblabes gnidiella</i> | Europe | |
| Coleoptera | | |
| <i>Craponius inaequalis</i> | Eastern United States | |
| <i>Lopus sulcatus</i> | France, Italy | |
| Heteroptera | | |
| <i>Euchistus conspersus</i> | California | |
| Chalcidoidea | | |
| <i>Prodecatoma cooki</i> | Florida | |
| Cecidomyiidae | | |
| <i>Contarinia viticola</i> | France | |
| Thysanoptera | | |
| <i>Drepanothrips reuteri</i> | North America, South Europe | |
| <i>Anaphothrips vitis</i> | Bulgaria, Romania, Greece, Turkey | |
| <i>Haplothrips globiceps</i> | Turkey | |
| <i>Retithrips aegyptiacus</i> | North Africa, Middle East | |
| <i>Rhipiphorothrips cruentatus</i> | India | |
| <i>Scirtothrips dorsalis</i> | Japan | |
| <i>Heliothrips haemorrhoidalis</i> | World | |
| <i>Frankliniella occidentalis</i> | California | |
| Honeydew producers | | |
| Hemiptera | | |
| <i>Planococcus citri</i> | South Europe | |
| <i>Pseudococcus maritimus</i> | California | |

(Continued)

Table 1 Grape insects of the world (*Continued*)

| Plant organ attacked | Pest | |
|----------------------------------|---|--------------------------|
| | Order | Region |
| Polyphagous insects | <i>Pseudococcus viburni</i> | California |
| | <i>Pseudococcus longispinus</i> | California |
| | <i>Planococcus ficus</i> | France, California |
| | <i>Parthenolecanium corni</i> | South Europe, California |
| | <i>Eulecanium persicae</i> | South Europe |
| | <i>Pulvinaria innumerabilis</i> | California |
| | <i>Pulvinaria vitis</i> | South Europe |
| | <i>Pulvinaria betulae</i> | Romania |
| | <i>Bemesia argentifolii</i> | California, Arizona |
| | <i>Trialeurodes vittatus</i> | California |
| | Orthoptera | |
| | <i>Melanopus devastator</i> | California |
| | <i>Oedaleonotus enigma</i> | California |
| | <i>Barbitistes fischeri</i> v. <i>berenguieri</i> | France, Spain, Italy |
| | <i>Ephippiger</i> spp. | South Europe |
| <i>Miogryllus convolutus</i> | South America | |
| <i>Locusta migratoria</i> | South Europe, North Africa | |
| <i>Schistocerca peregrine</i> | South Europe, North Africa | |
| <i>Doclostaurus maroccanus</i> | South France | |
| Coleoptera | | |
| <i>Macroductylus subspinosus</i> | Eastern United States | |
| <i>Popilia japonica</i> | Eastern United States | |
| <i>Anomala</i> spp. | France | |
| <i>Melolontha melolontha</i> | Europe | |
| <i>Polyphylla fullo</i> | Europe | |
| <i>Anoxia villosa</i> | Europe | |
| <i>Otiorrhynchus sulcatus</i> | Bulgaria, Western United States | |
| <i>Otiorrhynchus turca</i> | Bulgaria | |
| <i>Agriotes obscurus</i> | Romania | |
| Hymenoptera | | |
| Vespidae | World | |

(Reprinted with permission from the Annu. Rev. Entomol. 1977, 22(c) by Annual Reviews www.annualreviews.org).

BIOLOGICAL CONTROL IN CALIFORNIA VINEYARDS

Parasites, predators, and diseases play a key role in management of pests in California vineyards. Pests that are often managed by biological control include grape and variegated leafhoppers, grape mealybug, grape leaffolder, western grape leaf skeletonizer, and both Willamette mite and Pacific mite.

Small wasps in the family Mymaridae (primarily *Anagrus* spp.) parasitize eggs of both variegated and grape leafhopper and in wine and raisin grapes, often provide effective management. Even in table grapes, these small wasps often reduce leafhopper populations so that no further management is necessary.

Grape mealybug is parasitized by six species of wasps that have historically provided control of this key pest in California. The six parasitoids are *Acerophagus notativentris* (Girault), *Pseudaphycus angelicus* (Howard), *Zarhopalus corvinus* (Girault),

Anagrus subalbicornis (Girault), *Pseudaleptomastix squammullata* (Girault), and *Anagrus clauseni* Timberlake. Although common in coastal winegrape-growing areas, the predator, *Cryptolaemus montrouzieri* Mulsant, does not survive the hotter interior valley of California.

Grape leaffolder is a sporadic pest of grape throughout California. The wasp, *Bracon cushmani* (Muesbeck), lays multiple eggs on the leaffolder and when present during the first brood can eliminate the need for insecticide treatments.

Western grape leaf skeletonizer is parasitized by a wasp, *Apanteles harrisinae* Muesbeck, and a fly, *Ametadoria missella* (Wulp). However, the primary biological control agent is a granulosis virus that is passed from female to egg and also from male to female during mating. The virus infects the larvae and has been established throughout California; it was responsible for the drastic reduction in skeletonizer populations seen during the 1990s.

Non-chemical management of both Willamette and Pacific mite is predicated on the presence of the predator mite, *Galandromous occidentalis* (Nesbitt). If this predator is present on a sufficient number of infested leaves, spider mite populations will decline naturally unless disturbed by the use of broad-spectrum insecticides.

CULTURAL AND PHYSICAL PEST MANAGEMENT

Grape farmers use a variety of cultural pest management practices to better manage vineyard pests. Such practices are established for omnivorous leafroller, *Platynota stultana* Walsingham, grape phylloxera, *D. vitifoliae* (Fitch), and Pacific mite.

Removal and destruction, during the winter, of unharvested grapes is key to the management of wintering larvae of omnivorous leafroller.^[5] Prior to the use of preemergent herbicides for weed control, grape farmers used a technique known as “French plowing” whereby a spring-operated plow removed weeds in the vine row while not harming the vine. This procedure also physically destroyed grounded grape clusters in the process of eliminating weeds. As farmers move to herbicides, undestroyed grape clusters remain on the ground and harbor omnivorous leafroller.

The most effective management technique for grape phylloxera is the use of resistant rootstocks. Rootstocks such as St. George, 1613, Dog Ridge, and Teleki 5C have varying levels of resistance to grape phylloxera (*D. vitifoliae*). Each rootstock is uniquely adapted to specific growing conditions in the state.

The use of dusting sulfur to manage powdery mildew negatively impacts management of Pacific mite due to mortality of the western predator mite, *G. occidentalis*. Many grape growers are moving to the less toxic wettable form of sulfur to manage disease and help preserve the predator mite.^[7]

INSECTICIDES IN CALIFORNIA GRAPE IPM

The development of DDT and other broad-spectrum insecticides during the 1940s and 1950s greatly impacted the development of IPM in grapes. At a time when the number of entomologists was increasing, more reliance was placed on the development of insecticides and acaricides than on long-term pest management programs. Not until factors such as insecticide resistance, secondary pest outbreaks, and pest resurgence became known did entomologists begin looking at integration of pesticides into a more sustainable management program.^[8]

Within the last decade new insecticides and miticides have been developed that can truly be integrated

into pest management programs. Examples include various types of synthesized insect growth regulators (buprofezin, pyriproxifen, and tebufenozide), and chloronicotynyl insecticides. These new generation insecticides are useful for management of caterpillars, mealybugs, leafhoppers, scale, and mites. Also, the use of horticultural mineral oils and potassium fatty acid soaps have shown efficacy on soft-bodied pests. Although not truly an insecticide, the synthesis of various pheromone products aids both in monitoring and in mating disruption programs for pests such as omnivorous leafroller and vine mealybug. These products are remarkably non-toxic or short term in toxicity to beneficial arthropods. They have integrated very well into an effective and affordable IPM program. The use of broad-spectrum organophosphates and carbamates has dropped dramatically in California during the new millennium. Currently, these more broadly toxic products are used against pests where pesticides with reduced risk to the environment are not effective.

REGULATORY CONTROL

Regulatory management of pests has become important due to the ease of transport from one country to another, one state to another, or to different parts of the same state. The movement of vine mealybug into and throughout California occurred when infested grape nursery plants were moved into non-infested areas of the state. Currently, the California Department of Food and Agriculture has strict regulations governing the movement of both plant material and infested produce, and statewide monitoring programs for pests such as glassy-winged sharpshooter and vine mealybug. Such programs are important in slowing down movement and detecting early infestations of pests.

CONCLUSIONS

Integrated pest management programs for grape vary greatly throughout the world. However, the components of an IPM program include proper pest and beneficial arthropod identification, methods for monitoring pest populations, use of thresholds for application of insecticides, and the ability to integrate various pest management methods. Where IPM works best, the components of the system are continually refined and there is an active program to educate practitioners. Without the development and extension of this information IPM programs will stagnate and growers will continue to rely on insecticides alone. In the United States, land grant universities serve this function and are key to developing and providing information to farmers and the public.

REFERENCES

1. Winkler, A.J.; Cook, J.A.; Kliewer, W.M.; Lider, L.A. In *General Viticulture*, 2nd Ed.; University of California Press: Berkeley, 1974.
2. Bournier, A. Grape insects. *Ann. Rev. Entomol.* **1977**, *22*, 355–376.
3. Clavigero, F.X. *The History of (Lower) California*; Lake, S.E., Gray, A.A., Eds.; Stanford University Press: California, 1937.
4. <http://www.wineinstitute.org> (accessed November 2004).
5. Flaherty, D.L.; Christensen, L.P.; Lanini, W.T.; Marois, J.J.; Phillips, P.A.; Wilson, L.T. In *Grape Pest Management*, 2nd Ed.; U.C. D.A.N.R.: Oakland, 1992; 33 pp.
6. Bentley, W.J.; Zalom, F.G.; Granett, J.; Smith, R.J.; Varela, L.G.; Purcell, A.J. *UC IPM Guidelines: Grape*; U.C. A.N.R.: Oakland, 2004; 3448, 85 pp.
7. Hanna, R.; Zalom, F.G.; Wilson, L.T.; Leavitt, G.M. Sulfur can suppress mite predators in vineyards. *Cal. Agric.* **1997**, *51* (1), 19–21.
8. Stern, V.M.; Smith, R.F.; Van der Bosh, R.; Hagen, K.S. The integrated control concept. *Hilgardia* **1959**, *29*, 81–101.

Greenhouse Plant Pathogens

Margery Daughtrey

Department of Plant Pathology, Cornell University, Long Island Horticultural Research and Extension Center, Riverhead, New York, U.S.A.

INTRODUCTION

Disease management challenges continually occur in greenhouses, even though the environment may be carefully regulated and the pathogen inoculum may be reduced by sanitation practices. The greenhouse structure restricts the casual ingress of insect vectors of diseases, but greenhouse-adapted disease vectors sometimes proliferate and create epiphytotics in enclosed space. A common example is the efficient vectoring of *impatiens necrotic spot virus* (INSV) and *tomato spotted wilt virus* (TSWV) by the Western flower thrips, *Frankliniella occidentalis*.^[1] In addition, environmental conditions prevalent in greenhouse flower or vegetable crop culture are highly conducive to certain diseases. Disease problems due to foliar or root pathogens may escalate rapidly when conditions are favorable.

Greenhouse pathogens are sometimes placed under strong selection pressure for resistance development because of frequent, thorough sprays and the relative absence of gene exchange with other populations. *Botrytis*, downy mildews, and powdery mildews are especially likely to develop fungicide resistance when materials with single-site mode of action are employed in greenhouses.

New culture systems designed to save labor, conserve water, and reduce greenhouse runoff also influence the nature and spread of pathogens. Ebb-and-flow and flood-floor irrigation systems that recirculate the nutrient solution, for example, necessitate efficient and reliable disinfestation of return water.

MANAGING GREENHOUSE PATHOGENS

Pathogens may be introduced to the greenhouse on seeds, cuttings, or prefinished plants, or may be resident in organic debris, water reservoirs, weeds, or resident “pet” plants, or on benching or floors. New—sometimes exotic—pathogens may be brought into the greenhouse on cuttings of new crops. Clean stock production systems that utilize culture indexing and virus indexing provide an essential safeguard against systemic diseases for a number of greenhouse flower crops.

Managing diseases effectively in the greenhouse requires intensive scouting as well as a thorough and current knowledge of symptoms and appropriate responses. A brief discussion will be provided here, covering some of the pathogens most often associated with greenhouses: *Botrytis*, powdery mildews, bacteria, water molds (*Pythium* and *Phytophthora*), and viruses.

BOTRYTIS CINEREA

B. cinerea is the most ubiquitous greenhouse pathogen, causing Botrytis blight or gray mold on both vegetable and floral crops (Fig. 1).^[2] The symptoms it causes include flower and leaf spots, as well as stem cankers. Some of the most susceptible greenhouse crops include bacopa, cyclamen, exacum, fuchsia, geranium, lettuce, lisianthus, poinsettia, rose, snapdragon, and tomato, but virtually all crops are considered to be potential hosts. Several factors foster the development of *Botrytis* in greenhouses: tight plant spacing, high humidity, and water on plant surfaces. Free water is needed for the *Botrytis* spores to germinate and penetrate the plant. Flower tissues are an excellent substrate for *B. cinerea*, so any delay in shipping spring bedding plants increases the opportunity for losses to *Botrytis*, and cut flower crops are also vulnerable.

Cultural and environmental techniques are essential for Botrytis blight control. These include prompt removal of plant debris, watering early in the day to allow foliage to dry before nightfall, using fans to circulate the air, heating and ventilating at sunset to prevent condensation, and spacing plants adequately.^[3] Deleafing of greenhouse tomatoes should be done in the morning so that wounds dry before night.^[4] Fungicide applications should be supplementary to other integrated pest management techniques, as fungicides used alone will not be effective in a highly disease-conducive environment.^[3]

In greenhouse flower production, the active ingredients chlorothalonil, fenhexamid, and fludioxonil are currently widely used. Vinclozolin and iprodione are also helpful, even though partial resistance to the dicarboximides has been documented.^[5] Mancozeb, coppers, and strobilurins also provide some *Botrytis* control. Resistance to the widely used benzimidazole



Fig. 1 The disease caused by *B. cinerea* is called gray mold because of the gray bloom of sporulation on lesions, shown here on a geranium leaf. Round spots or wedges of necrosis are typical on leaves.

fungicides (benomyl and thiophanate-methyl) has been repeatedly documented in greenhouse vegetable and flower crops,^[5] so that this chemistry is no longer considered helpful for managing *Botrytis*. Biological controls are not widely used to manage this disease.

POWDERY MILDEWS

Although there are many different powdery mildew species, most of these fungi form similar-looking whitish colonies on the surface of their plant hosts (Fig. 2). Many powdery mildews are well adapted to the greenhouse environment. Some of the crops especially vulnerable to powdery mildew are African violet, begonia, gerbera, hydrangea, poinsettia, rose, and verbena, as well as greenhouse cucumber and tomato. Since 1990, an *Oidium* sp. has been a threat to poinsettia production in North America. This powdery mildew requires temperatures below 85°F for development,^[6] so it may go unnoticed until the final months of poinsettia production. A new powdery mildew of



Fig. 2 Powdery mildew fungi often form conspicuous white colonies on leaves, as seen here on gerbera.

tomatoes (*Oidium neolycopersici*) has also been problematic in the past decade.^[7]

Environmental control alone is not usually sufficiently effective against powdery mildews. Scouting is important for effective management, so that chemical control programs may be initiated only when inoculum is present and the environment favors disease. Roguing out heavily infested plants reduces inoculum. Fungicides for powdery mildew control include the contact materials piperalin and potassium bicarbonate, as well as ergosterol biosynthesis inhibitors and strobilurins. Mildew-tolerant cultivars of cucumber and tomato are used for greenhouse culture, at times supplemented with fungicides.^[4] Programs rotating between materials with different modes of action should be utilized to slow down the development of resistance. Recently, biological materials including *Bacillus subtilis* and *Pseudozyma flocculosa*^[8] have been developed to counter powdery mildew.

BACTERIA

Bacteria prosper in temperatures and humidities typical of greenhouses, and are easily spread by handling or splashing. Bacterial pathogens are sometimes introduced on seed, or in latently infected plant material. Diseases caused by bacteria are commonly cited by greenhouse growers as the most difficult to manage category, particularly because chemical tools are limited to copper materials.

The most common bacterial diseases in greenhouses are leaf spots caused by *Xanthomonas* and *Pseudomonas* spp., as well as stem and corm rots due to *Erwinia* species. The most dangerous pathogens are those causing vascular wilts. These include *Xanthomonas campestris* pv. *pelargonii* (Fig. 3) and *Ralstonia solanacearum*.^[2]

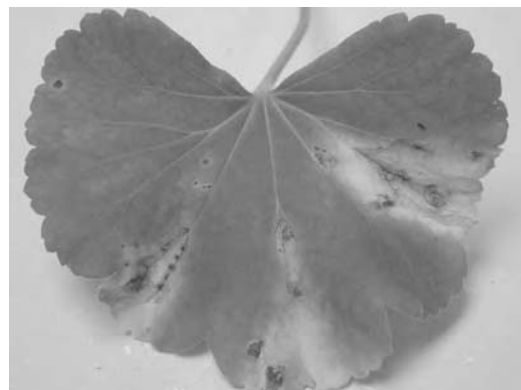


Fig. 3 *X. campestris* pv. *pelargonii* causes tiny, round, necrotic spots as well as necrotic wedges on infected geranium leaves. There may also be chlorosis around the lesions.

Sanitation and exclusion are the mainstays of bacterial disease control. Biocontrol with *B. subtilis* has recently become an option. Soft rots (*Erwinia* spp.) are controlled largely through sanitation and avoidance of stress to cuttings during propagation. Systemic diseases are managed by clean stock production employing culture indexing, coupled with prompt eradication if these disease exclusion programs are inadvertently compromised. The recent introduction of Race 3, Biovar 2 of *R. solanacearum* into Europe, the United States, and Canada via geranium cuttings from Kenya and Guatemala has led to increased regulatory scrutiny and a tightening of sanitation protocols for all geranium production facilities.

WATER MOLDS (*PYTHIUM* AND *PHYTOPHTHORA*)

Root and stem diseases in greenhouse flower culture are most commonly caused by oomycetes (“water molds”). *Pythium* species are most often associated with root rot (Fig. 4),^[9] whereas crown and stem rots are more typical for *Phytophthora*.^[10] Sanitation is a key element of control programs. A growing medium with high porosity is needed, and excessive nitrogen should be avoided.^[11] Fungus gnats can vector water molds,^[12] so these must also be managed. Production in pots or individual bags of soilless mix separates plants’ root systems and curtails pathogen spread. In contrast, recirculating subirrigation of plants, increasingly used for ornamental crops, presents an immense potential for dissemination of *Pythium* and *Phytophthora*. In recognition of this danger, operations with recirculating irrigation usually use some method of filtration, coupled with water treatment via ultraviolet light, chlorine, or peroxide.



Fig. 4 Yellowing of lower foliage and wilting will be observed on poinsettias with root rot caused by *Pythium aphanidermatum*, which is active at summer greenhouse temperatures.

Fungicides are sometimes needed for *Pythium* control, especially with highly susceptible crops such as poinsettia, geranium, and calibrachoa. *Phytophthora* diseases may require chemical treatment in fuchsia, gloxinia, gerbera, calibrachoa, pansy, and poinsettia. Although, until recently, metalaxyl and mefenoxam have been the primary materials used for water mold control, over half of greenhouse flower crop isolates of *Pythium* in the Northeast have recently been shown to be insensitive to this chemistry.^[9,13] The alternative options for water mold control in ornamentals include etridiazole, fosetyl-Al, and new phosphorous acid materials. Biological controls are increasingly being used for root rot suppression.

VIRUSES

During the past two decades, the primary greenhouse virus problem has been INSV, vectored by Western flower thrips^[1] (Fig. 5). This virus has a wide host range including flowers, vegetables, and weeds, some of which are non-symptomatic. Greenhouse tomato crops are very susceptible to TSWV. Should the Western flower thrips become resistant to the chemistry currently being used for its control, there could be a quick resurgence of both INSV and TSWV in greenhouses. Management of INSV and TSWV is dependent on the recognition of a bewildering array of symptoms so that diseased plants can be rogued out promptly. The thrips vector population must be monitored, and its population and that of reservoir weeds must be kept under tight control.

Other major virus problems on greenhouse tomatoes are *tomato mosaic virus* and *pepino mosaic*. Significant ornamental virus losses have recently been due to *tobacco mosaic virus* in petunia and other



Fig. 5 Black lesions are sometimes formed on New Guinea impatiens or garden impatiens infected by INSV.

vegetatively grown annuals. A number of unidentified viruses are encountered each year, especially on newly introduced ornamental crops.

CONCLUSION

Profitable greenhouse production requires expertise in sanitation practices and skillful management of the environment to avoid disease-conducive conditions. Future research should focus on the technology for producing seed and cuttings free from pathogens, so that only clean plant material is brought into the greenhouse. Refinements in pathogen detection and in the art of culture indexing and virus indexing are needed to improve the biosecurity of greenhouse crops.

REFERENCES

1. Daughtrey, M.L.; Jones, R.K.; Moyer, J.W.; Daub, M.E.; Baker, J.R. Tospoviruses strike the greenhouse industry: INSV has become a major pathogen on flower crops. *Plant Dis.* **1997**, *81*, 1220–1230.
2. Daughtrey, M.L.; Wick, R.L.; Peterson, J.L. *Compendium of Flowering Potted Plant Diseases*; APS Press: St. Paul, MN, 1995.
3. Hausbeck, M.K.; Moorman, G.L. Managing *Botrytis* in greenhouse-grown flower crops. *Plant Dis.* **1996**, *80*, 1212–1219.
4. Anonymous. In *Growing Greenhouse Vegetables*; Ontario Ministry of Agriculture, Food and Rural Affairs, Publication 371, 2001.
5. Yourman, L.F.; Jeffers, S.N. Resistance to benzimidazole and dicarboximide fungicides in greenhouse isolates of *Botrytis cinerea*. *Plant Dis.* **1999**, *83*, 569–575.
6. Byrne, J.M.; Hausbeck, M.K.; Shaw, B.D. Factors affecting airborne concentrations of conidia of *Oidium* sp. among poinsettias in a greenhouse. *Plant Dis.* **2000**, *84*, 1089–1095.
7. Jones, H.; Whipps, J.M.; Gurr, S.J. The tomato powdery mildew fungus *Oidium neolycopersici*. *Mol. Plant Pathol.* **2001**, *2* (6), 303–309.
8. Dik, A.J.; Verhaar, M.A.; Belanger, R.R. Comparison of three biocontrol agents against cucumber powdery mildew (*Sphaerotheca fuliginea*) in semi-commercial scale glasshouse trials. *Eur. J. Plant Pathol.* **1998**, *104*, 413–423.
9. Moorman, G.W.; Kang, S.; Geiser, D.M. Identification and characterization of *Pythium* species associated with greenhouse floral crops in Pennsylvania. *Plant Dis.* **2002**, *86*, 1227–1231.
10. Beckett, M.; Daughtrey, M. *Scary Phytophthoras*; Greenhouse Grower: September 2002; 90–92.
11. Moorman, G.W. Increased plant mortality caused by *Pythium* root rot of poinsettia associated with high fertilization rates. *Plant Dis.* **1986**, *70*, 160–162.
12. Jarvis, W.R.; Shipp, J.L.; Gardiner, R.B. Transmission of *Pythium aphanidermatum* to greenhouse cucumber by the fungus gnat *Bradysia impatiens* (Diptera, Sciaridae). *Ann. Appl. Biol.* **1993**, *122*, 23–29.
13. Shishkoff, N.; Knoedler, J.; Daughtrey, M. Sensitivity to propamocarb and mefenoxam in *Pythium* isolated from within flower crop greenhouses. *Phytopathology* **2003**, *93* (6), S135.

Groundwater, Pesticides in

Walter Brüsch

*Department of Geochemistry, Geological Survey of Denmark and Greenland (GEUS),
Copenhagen K., Denmark*

Gitte Felding

Geological Survey of Denmark and Greenland (GEUS), Copenhagen K., Denmark

INTRODUCTION

Results from monitoring systems show that the use of pesticides in urban and agricultural areas influences surface and groundwater.^[1-3] When pesticides are used in urban areas, e.g., on roads and pavements, there is a risk of infiltration of pesticides because of lack of the biological active root zone. The pesticides will degrade slower, and often the sorption (K_d) will be very low in the topsoil, compared with normal agricultural root zones. In agricultural areas, there is an increased risk from point sources^[4] and also for infiltration of pesticides and metabolites when pesticides are used prior to heavy rain^[5,6] or in the autumn. The soil type also plays an important role, for example, if pesticides are used on fractured clayey soil types (tills) with preferential flow through fractures, sand lenses, root channels, or wormholes.

In Denmark, detection of pesticides in small waterworks is common when groundwater is extracted from reservoirs near the surface. Pesticides can also contaminate groundwater from deeper reservoirs where well construction and use of herbicides near the well often can be blamed.

It should be expected that pesticides with strong sorption (high K_{oc}) and a low half-life (DT_{50}) remain and degrade in the root zone.^[7] However, many cases have shown that findings of the presence of pesticides in groundwater often can be explained by unusual events. Therefore, even pesticides that are not “possible leachers” can be found in the groundwater^[5] if they are used frequently enough and in large quantities.

PESTICIDES FOUND IN MONITORING PROGRAMS IN EUROPE AND THE USA

Information about more than 550 pesticides (and their metabolites) used in Denmark from 1956 to 1998 has been collected. The results from 50 different monitoring and investigations programs from Europe and the USA have been processed in a database and the findings evaluated.^[8] Approximately 300 pesticides and

metabolites have been analyzed and 140 have been found. A minor number of substances are only reported as “found”^[9] and no information about number of analyses or circumstances were reported.

In monitoring programs where only few parameters are analyzed, it is normal to find all compounds, whereas in large programs, it is common to find only some of the analyzed parameters. However, a trend is increasing number of parameters → increasing number of parameters found (Fig. 1). Other obvious limiting factors could be detection limits, well type, analytic methods, areal use (agricultural, urban, roads, or railways), and monitoring purpose.

Table 1 compares pesticides and metabolites detected in monitoring programs in the USA, Europe, and Denmark. Only frequently analyzed pesticides have been included:

- Pesticides analyzed more than 100 times in monitoring programs.
- Pesticides analyzed more than 200 times in larger compiled programs.
- Pesticides analyzed in more than 2–3 programs.

Table 1 shows that 2,6-dichlorbenzamide (BAM) is found frequently in Denmark, whereas atrazine and metabolites are detected most frequently in Europe and the USA. BAM has often been found in urban areas and not in young groundwater samples from agricultural areas. In Europe, bentazone, simazine, diuron, isoproturon, and two phenoxy acids have also been frequently detected. Ethylene thiourea (ETU) has been found in Denmark, but it should be noted that the detection originates from groundwater sampled in selected wells and that the analytic method is difficult.

DANISH GROUNDWATER MONITORING PROGRAM

Groundwater monitoring was initiated in 1987 to 1990 to assure the quality of drinking water and to demonstrate the effect of political actions against

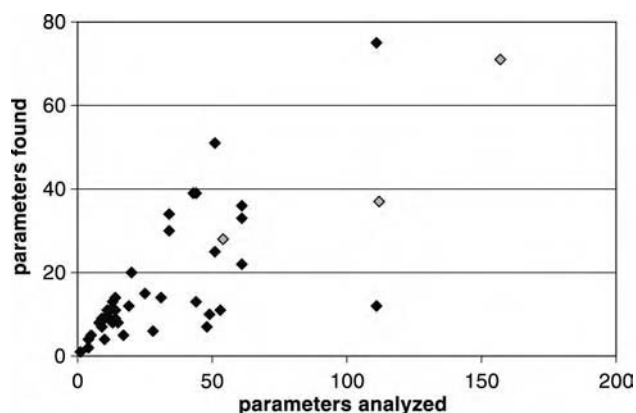


Fig. 1 Number of pesticides and metabolites analyzed and number of parameters detected in 47 monitoring programs from Europe and the United States. Three data sets from Denmark are marked with gray marks. (From Ref.^[8])

groundwater pollution. The analytical program included nitrate, chloride, dissolved iron, inorganic trace elements, pesticides, and other organic micropollutants.

The local counties are responsible for the Danish National Monitoring Program and report once a year analytic data to the Geological Survey of Denmark and Greenland (GEUS).^[2,10,11] The monitoring system^[12] is based on analysis of groundwater samples from 67 monitoring areas, five agricultural watersheds, and of drinking water by the waterworks.^[13]

The groundwater monitoring areas consist of one abstraction well to drive the groundwater flow and

15 to 20 monitoring wells in the main aquifer and in the secondary aquifers,^[2,14] in total approximately 1100 well screens.

In the five agricultural watersheds, 100 shallow wells (1.5- to 5-m depth) are analyzed up to four times a year to elucidate the situation in groundwater below farmland.^[15]

The groundwater monitoring program also includes analysis of samples from waterworks taken at least once every 3 to 5 years in approximately 10,000 wells, depending on the size of the water supply.^[13]

CHEMICAL PROGRAM

Based on the groundwater monitoring results up to 1997, the program was revised and the latest program started in 1998.^[12] Description of the monitoring program can also be found on the GEUS water resource website.^[16]

The program includes 21 main chemical elements, 4 field measurements, 23 inorganic trace elements, 21 organic micropollutants, and 46 pesticides and metabolites. The predominant detection limit is 0.01 µg/L, when analyzing pesticides and metabolites.

Sampling frequencies are generally once a year in young groundwater, and less often in old groundwater. Age dating of the groundwater is based on tritium and CFC isotope analysis.^[2]

The groundwater quality monitoring results are described in annually monitoring reports published by GEUS and presented in the GEUS website.^[17]

Table 1 The 10 most frequently found pesticides and metabolites in groundwater in USA, Europe, and Denmark

| USA | | Europe | | Denmark, national monitoring system | | Denmark, all analytical programs | |
|-----------------|----------------|---------------------|----------------|-------------------------------------|----------------|----------------------------------|----------------|
| Pesticide | Avg. frequency | Pesticide | Avg. frequency | Pesticide | Avg. frequency | Pesticide | Avg. frequency |
| Atrazine | 1.3 | Atrazine | 2 | BAM | 1 | BAM | 1 |
| Deethylatrazine | 2.5 | Deethylatrazine | 2 | Deethylatrazine | 4.3 | Deethyldeisopropylatrazine | 2 |
| Simazine | 3 | BAM | 2.5 | Deisopropylatrazine | 4.3 | Deethylatrazine | 6 |
| Prometon | 3.8 | Bentazone | 4 | Atrazine | 5 | Deisopropylatrazine | 7 |
| Metolachlor | 5 | Simazine | 5.3 | Bentazone | 6.7 | Bentazone | 8 |
| Tebuthiuron | 6.5 | Diuron | 5.5 | Mecoprop | 7.7 | Atrazine | 9 |
| Alachlor | 8.3 | Isoproturone | 6.5 | Dichlorprop | 8 | Simazine | 12 |
| Carbofuran | 9.3 | Deisopropylatrazine | 7 | MCPA | 8 | Dichlorprop | 13 |
| Cyanazine | 9.3 | Mecoprop | 7.7 | Simazine | 9.5 | ETU | 14 |
| Metribuzin | 10.3 | Dichlorprop | 8 | Hydroxyatrazine | 10 | Mecoprop | 15 |

A low average frequency number indicates that the substance has been found most frequently in the monitoring programs used as background material. A top 10 list for the individual program has been calculated. Summing up all top 10s and dividing by the number of programs gives an average frequency. For example, BAM has a value "1" in the column "Denmark, all analytic programs," indicating that BAM was detected most frequently in all the programs.

Table 2 Pesticides and metabolites found in Danish groundwater, 1993–1999

| Pesticides/metabolites | Number of analyzed wells | Wells with findings | | Findings $\geq 0.1 \mu\text{g/L}$ | |
|-------------------------------|--------------------------|---------------------|------|-----------------------------------|------|
| | | Number | % | Number | % |
| GRUMO ^a | 1.061 | 371 | 35.0 | 114 | 10.7 |
| LOOP ^b | 119 | 63 | 52.9 | 20 | 16.8 |
| Extraction wells ^c | 5.774 | 1.396 | 24.2 | 509 | 8.8 |

^aNational Monitoring System.

^bNational monitoring of young groundwater under agricultural areas.

^cMonitoring of extraction wells carried out by the Danish waterworks, only wells with extraction of groundwater in the period 1996–1999 included. EU-MAC = $0.1 \mu\text{g/L}$.

From Ref.^[2].

PESTICIDES AND METABOLITES FOUND IN DANISH GROUNDWATER

Table 2 shows that analytic results from the monitoring systems in Denmark differ. In water samples collected from young groundwater in agricultural areas, only pesticides used on the fields were found in the groundwater samples. In water samples collected in the National Groundwater Monitoring System (GRUMO), pesticides used in urban areas were also found.

The waterworks analyze water samples from extraction wells and the analytical results show a significant dominance of BAM, a metabolite from dichlobenil.

From 1993 to 1999, pesticides and metabolites have been found in about 35% of the monitoring wells in the National Monitoring System, 12% above the European Union maximum concentration level (EU-MAC) ($0.1 \mu\text{g/L}$). The corresponding values for 1999 are 24% and 7%, respectively (Fig. 2 and Table 2).

Detection of pesticides in the monitoring system decreases with increasing depth, but pesticides/metabolites have been found at depths greater than 100 m (Fig. 3). Pesticides and metabolites influenced more than 50% of the sampled monitoring screens 0–10 m

below surface. The most frequently found groups of pesticides and metabolites were triazines, phenoxy acids (including metabolites), and BAM.

In shallow young groundwater (LOOP), pesticides and metabolites have been found in 53% of the investigated monitoring wells where the EU-MAC level was exceeded by 17%. The most frequently detected pesticides are triazines, phenoxy acids, and glyphosate. Glyphosate and its metabolite aminomethylphosphonic acid (AMPA) have been found in drain and young groundwater, where preferential flow in fractures and root channels in an unweathered glacial till deposit have transported glyphosate and AMPA at least 1 m below the surface after rainstorms. Glyphosates were used on stubble fields after harvest.^[5] Glyphosate was also detected in groundwater 5 m below the surface, but at this depth, transport through leaky wells may be the reason, although tracer test indicated that preferential flow along macropores and fractures dominate the horizontal and vertical transport of water. In such fractured glacial tills, degradation and sorption of glyphosate may be minimal because of the rapid transport rates and short residence times. This implies that the glacial till offers minimal protection to surface water and to the underlying aquifers.

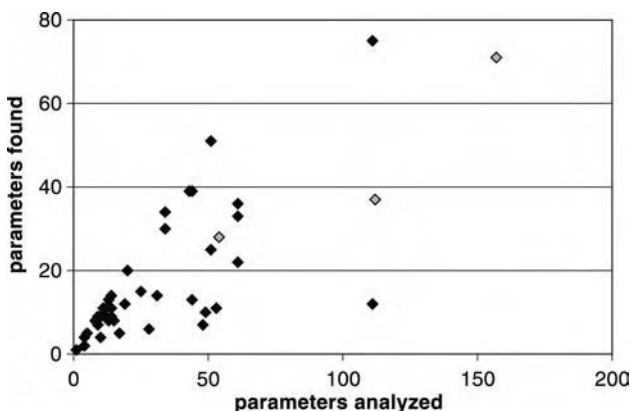


Fig. 2 Pesticides and metabolites found in groundwater during 1993 to 1999. The National Monitoring System. (From Ref.^[2].)

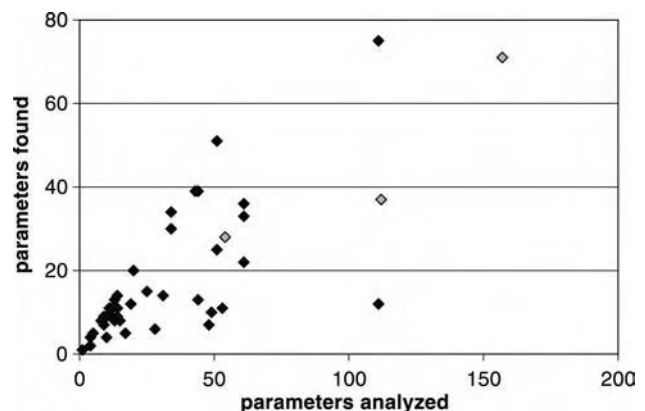


Fig. 3 Occurrence of pesticides and metabolites in relation to depth below surface in 1999. The National Monitoring System. (From Ref.^[2].)

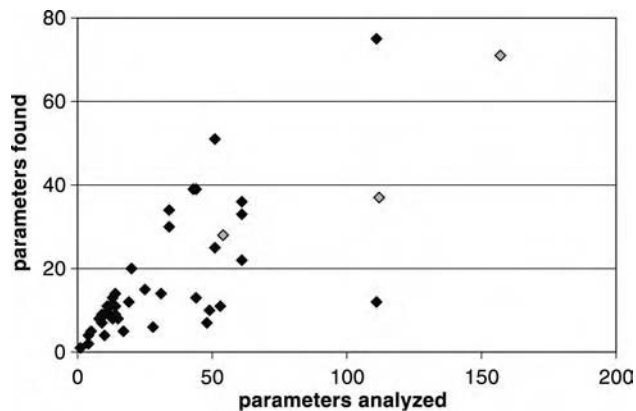


Fig. 4 Nitrate concentration in milligrams per liter and BAM, atrazine, dichlorprop, and mecoprop (MCP) concentration in micrograms per liter. The phenoxy acids, dichlorprop, and mechlorprop dominate in anaerobic groundwater with <1 mg O_2 and <1 mg nitrate whereas atrazine and BAM also occur in aerobic groundwater.

Detection of pesticides in different redox environments shows an overrepresentation of pesticides in aerobic environments (Fig. 4), which could indicate that some pesticides degrade under anaerobic conditions, or more likely that pesticides are biodegraded under transport through young and aerobic groundwater environments. On the other hand, specific groups of pesticides occur especially in anaerobic environments, presumably indicating that, e.g., phenoxy acids are degraded in aerobic environments, but are stable when transported to anaerobic groundwater. Phenoxy acids are detected in anaerobic sand reservoirs under bedded till and clay layers.

Monitoring results from extraction wells showed pesticides and metabolites in 24% of the analyzed wells (Table 2). BAM was found in 24% of 4202 analyzed water supply wells, and the EU-MAC level (0.1 $\mu\text{g/L}$) was exceeded by 10%. BAM is frequently found in wells located in urban areas, along roads and railways, and on farmyards. Triazines and their metabolites make up another frequently found group. These compounds are commonly found in farming areas, but are also found in urban areas. In agricultural watersheds, the triazines and their metabolites make up almost half of all pesticides and metabolites detected. Forty-seven pesticides and metabolites have been detected in monitoring wells, whereas 87 have been found in Danish groundwater.^[2]

CONCLUSION

Monitoring of pesticides and their metabolites in US and in Europe show that use of agricultural chemicals

cause significant impact on ground- and drinking water. Even pesticides with strong sorption and low half-life can be found in many types of groundwater reservoirs. Worldwide, the most frequently found pesticides and metabolites belong to the triazine group. In Denmark the most frequently found metabolite is 2,6-dichlorbenzamide (BAM). BAM has been found in 24% of the Danish water supply wells and causes many problems with the drinking water quality.

REFERENCES

1. Jacobsen, C.S.; Brüsich, W.; Lindhardt, B. *Evaluation: Leaching of Pesticides From Roadways and Other Non Agricultural Areas (Vurdering af Risiko for Udvaskning af Pesticider fra Befæstede Arealer)*. Danmarks og Grønlands Geologiske Undersøgelse, Report 55; The Geological Survey of Denmark and Greenland, 1998.
2. *GEUS 2000: Groundwater Monitoring 2000 (Grundvandsovervågning 2000)—GEUS Special Edition*; The Geological Survey of Denmark and Greenland, 2000. www.grundvandsovervaagning.dk.
3. Jacobsen, O.S.; Brüsich, W.; Fomsgaard, I.; Helweg, A.; Vinther, F.P.; Spliid, N.H.; Bossi, R.; Mogensen, B.B. *Leaching and Degradation of Selected Pesticides in a Sandy Agricultural Watershed. Bekæmpelsesmiddel Forskning fra Miljøstyrelsen. Report No. 46, Miljøstyrelsen*; The Danish Environmental Protection Agency, 1998.
4. Helweg, A.; Brüsich, W.; Jacobsen, O.S.; Spliid, N.H.; Hansen, S.U.; Laier, T. *Pesticides in Point Sources. The Origin and Spreading in Soil and Water (Pesticider i Punktkilder. Punktkildernes Oprindelse og Spredning i Jord og Grundvand)*. Bekæmpelsesmiddelforskning fra Miljøstyrelsen; The Danish Environmental Protection Agency, 1999. www.mst.dk/199909publikat/87-7909-335-3/helepubl.doc.
5. Nilsson, B.; Brüsich, W.; Morthorst, J.; Vosgeraul, H.; Abildtrup, C.; Pedersen, D.; Jensen, P.; Clausen, E.V. *Investigation of LOOP Wells DGUnr 165.295–165.297 in LOOP Ares No. 4. (Undersøgelse af Landovervågningsboringerne DGUnr. 165. 295–165 297 i LOOP Område 4, Lillebæk, Fyns amt.) GEUS Report No. 47*; The Geological Survey of Denmark and Greenland, 2000.
6. Nilsson, B.; Harrar, W.; Brüsich, W.; Vosgerau, H.; Abildtrup, C.; Morthorst, J.; Bojesen, O. *Glyphosate Leaching to Tile Drains and Shallow Groundwater in a Macroporous Glacial Till*, International Conference "Fractured Rock 2001," Toronto, Ontario, Canada, March, 26–28, 2001.
7. Lindhardt, B.; Fomsgaard, I.S.; Brüsich, W.; Bossi, R. *Pesticiders Udvaskelighed—Vurdering af Usikkerheden på DT_{50} og K_{oc} , der Anvendes i GUS-indekset. Arbejdsrapport fra Miljøstyrelsen*; The Danish Environmental Protection Agency, 2000. <http://www.mst.dk/udgiv/publikationer/2000/87-7909-586-0/html>.

8. Brüsch, W.; Felding, G. *Pesticides in Danish and Foreign Ground Water (Pesticider i Dansk og Udenlandsk Grundvand)*. "State of the Art" Project—GEUS Report No. 105; The Geological Survey of Denmark and Greenland, 2000.
9. Heinz, I.; Flessua, A.; Zullei-Seibert, N.; Kuhlmann, B.; Schulte-Ebert, U.; Michels, M.; Simbrey, J.; Fleischer, G. *Economic Efficiency Calculations in Conjunction with the Drinking Water Directive (Directive 80/778/EEC). Part III: The Parameter for Pesticides and Related Products. Final Report for the European Commission—DGXI. Contract no. B4-3040/94/000223/MAR/BI*; The European Commission, 1995.
10. GEUS. *Groundwater Monitoring 1998 (Grundvandsovervågning 1998)—GEUS Special Edition*; The Geological Survey of Denmark and Greenland, 1998. www.grundvandsovervaagning.dk.
11. GEUS. *Groundwater Monitoring 1999 (Grundvandsovervågning 1999)—GEUS Special Edition*; The Geological Survey of Denmark and Greenland, 1999. www.grundvandsovervaagning.dk.
12. Miljøstyrelsen NOVA-2003. *Programbeskrivelse for det nationale program for overvågning af vandmiljøet 1998–2003—Statements from the Danish EPA*; 2000. http://ovs.dmu.dk/2NOVA_2003_ov./novaarkivet/NOVA-program-version4.doc.
13. Miljøministeriet *Bekendtgørelse fra Miljøministeriet nr. 515 af 29. August 1988, om Vandkvalitet og Tilsyn med Vandforsyningsanlæg*; Environmental Ministry of Denmark, 1988.
14. Czako, T. Groundwater monitoring network in Denmark: example of results in the Nyborg area. *Hydrol. Sci. J.* **1993**, 39 (1), 1–17.
15. Rasmussen, P. Monitoring shallow groundwater quality in agricultural watersheds in Denmark. *Environ. Geol.* **1996**, 27, 309–319.
16. www.groundwater.dk.
17. www.grundvandsovervaagning.dk.

Hazard Labeling

Leslie London

Occupational and Environmental Health Research Unit, School of Public Health and Primary Health Care, University of Cape Town, Observatory, Western Cape, South Africa

Hanna Andrea Rother

Occupational and Environmental Health Research Unit, School of Public Health and Family Medicine, University of Cape Town, Observatory, South Africa

INTRODUCTION

Hazard communication tools, including labeling and Safety Data Sheets (SDS), play a key part in preventing the adverse impacts of pesticides on human health and the environment. Hazard communication potentially plays one of two main roles: to provide chemical hazard information, and to motivate cautionary behavior.^[1] In many countries, pesticide labels are part of regulatory controls, and therefore also serve to extend legal liability to the producer, end user, or both. However, hazard communication tools may perform poorly outside those settings where such strategies were developed, and the effectiveness of hazard communication, particularly in developing countries, is often assumed without empirical evidence of efficacy.

PESTICIDE LABEL COMPONENTS

Pesticide labels are intended to provide the first alert that a chemical is potentially hazardous and covers basic information on safe handling, dosage, protective measures, emergency first aid, the pesticide's hazards, as well as the identity of the active ingredient, and the producer name and contact details.

Signal Words

Signal words are single words on a pesticide label intended to get the reader's attention (e.g., warning, toxic, and danger). Their effectiveness is increased if the signal words precede an action statement.^[2] However, the ability of signal words to do more than draw attention to a hazard statement (e.g., distinguishing the hazard level or nature of the risk) is limited. Numerous studies show that consumers and users fail to distinguish consistently between terms such as "caution" and "warning."^[2]

Symbols

Symbols (e.g., pictograms) aim to convey a physical or biological hazard associated with a pesticide, with

the assumptions that they overcome language barriers, provide quick communication, use little space, and attract attention from a distance. Pictograms, developed jointly by the Food and Agriculture Organization (FAO) and the pesticide industry, have formed the mainstay of pesticide hazard communication, particularly in developing countries (see Fig. 1). Pesticide pictograms indicate activity (e.g., spraying, mixing, and storage), advice, and warnings. By 1988, the pesticide industry and the FAO began to promote pictograms for use and, by 1990, 69% of all countries were reported to be using them.^[3]

However, although symbols may attract attention, this is no guarantee of understanding. There is extensive evidence that many symbols are not well understood, even among populations with high levels of literacy.^[2]

Colors

Colors are used to reinforce the effectiveness of hazard symbols, and literature suggests some consistency in the hierarchy of red–purple–orange–yellow–blue–green–white reflecting a toxicity ranking.^[4] However, findings of consistency in colors were not supported in a study among workers in Zimbabwe,^[5] and the cross-cultural applicability of color codes has had little empirical testing in developing countries.

HOW COMPREHENSIBLE CAN WE EXPECT PESTICIDE LABELS TO BE?

Attributes of the Message

Message attributes influencing readability include font size, line length and type size, contrast and meaningful segmentation of the text, paragraph justification, and avoidance of italics to improve legibility. Hazard symbols seem most effective when located at the top right corner of a label.

Increasing the explicitness of label precaution information increases both instruction reading and
















| Crop Life International (GIFAP) / Food and Agricultural Organisation (FAO) Pictograms. | | | |
|--|---|---|-----------------------------|
| Advice & Warning Pictogram: | Indicates | Activity Pictogram: | Indicates |
|  | Wear gloves |  | Handling liquid concentrate |
|  | Keep locked away and out of reach of children |  | Handling dry concentrate |
|  | Wash after use |  | Application |
|  | Wear protection over nose and mouth | | |
|  | Wear boots | | |
|  | Dangerous/harmful to wildlife and birds | | |
|  | Not for aerial application | | |
|  | Wear respirator | | |
|  | Dangerous/harmful to fish and water bodies | | |
|  | Dangerous/harmful to livestock and poultry | | |
|  | Wear eye protection | | |
|  | Expiry date | | |

Fig. 1 Hazard communication symbols for pesticide labels. (Adapted from Crop Life International, *Pictograms for Agrochemical Labels—An Aid to Safe Handling of Pesticides*, GCPF, Brussels, 1988; revisions and additions, 1993.)

compliance among consumers. The type of information is also important for ease of comprehension. For example, information that relates to procedures (e.g., first aid) is easier understood than more complex health risks (e.g., chronic effects). Research suggests that, in western societies, hazard comprehension increases proportionally to the perceived severity of the hazard.

Studies also suggest that subjects read warnings more often if they precede use instructions, but others have found better comprehension with the integration of precautionary statements with use instructions. However, much of this research stems from studies with university students or consumers, and has little relevance to populations in developing countries where pesticide exposures are highest.

Attributes of the Reader

Gender difference in perception and behavior has been widely demonstrated across cultures, with women more likely to both read and comply with warnings. Some pieces of evidence suggest that younger workers tend to notice warnings and have better understanding,

but that older workers tend to act upon warnings once noticed.^[2]

Higher levels of formal education are expected to increase levels of hazard labeling understanding and may confound age-related findings. However, adult education research findings show that workers with little formal education, but who have life experience in particular fields, develop knowledge and insights that increase their skills and comprehension of messages, including those related to hazards and safety in their working environment.

In addition, visual literacy is critical for the interpretation of graphics. Formal schooling promotes visual literacy, and enables the learner to make sense of stylized and complex graphics, and to process both familiar and unfamiliar items. Experience with poorly educated farm workers^[2] suggests that they preferred realistic images (preferably photographs) rather than representational images because they related to their real experience. Therefore pictograms, safety symbols, and other graphic messages all rely on some level of a priori (tacit) knowledge. Hence “no hazard communication system is intuitively obvious,”^[6] and the role of social learning in interpreting hazard symbols and messages is emphasized in the literature.^[1]

Training plays a critical role in hazard communication effectiveness. Even brief explanations of meaning appear to play a vital role in improving comprehension.^[4] The meaning of symbols, colors, and all other label elements should be taught to be understood, beginning with primary education.

Attributes of the Environment

Consumer research suggests that product familiarity decreases users' reading of instructions, reducing effective hazard communication. In contrast, work among Kenyan women suggested the opposite—that product familiarity and prior experience potentially increased comprehension of, and compliance with, pharmaceutical instructions for oral rehydration therapy.^[2] In visual languages, prior experience and prior knowledge influence comprehension. Differences in the cultural specificities of responses to hazard warnings may explain these apparent differences between consumers



Fig. 2 European Union oxidizing symbol.

and developed world workers, and Kenyan women. Moreover, even if familiarity reduces the “effectiveness” of a warning, it may not necessarily lead to reduced compliance because the precautionary action may be as familiar as the warning.^[2]

Nonetheless, it is clear that the message context is key. For workers in developing countries, this may pose a threat to the validity of the hazard communication icons presumed to be universally understood. It is thus not surprising that Baloyi^[5] reported Malawian workers identifying the European Union (EU) symbol for oxidizing agent as a germinating flower (see Fig. 2).

HAZARD COMPREHENSION

To convey to users key information that influences safety-promoting behavior, three levels of impact should be considered:

- Cognitive level—information provision, message perception, awareness, knowledge, and understanding.
- Emotive level—identification of personal risk and risk to others, and weighing of such risk (perceptions and attitudes).
- Behavioral level—safe practices implementation on individual or collective levels, which is the essential objective of hazard communication.

Hazard communication must be seen as an adjunct to training, engineering controls, and other measures for promoting chemical safety. An exclusive focus on labels and SDS runs the risk of shifting the responsibility for pesticide safety to the individual worker or consumer. Labels and SDS must form part of an integrated program for improving the safe management of pesticides and not as a substitute for safe design, good safety practice, and policies to reduce pesticide exposures.

Table 1 Comprehensibility of hazard communication symbols among 222 Ethiopian workers

| | |
|----------------------|-----|
| Toxic | 7% |
| Corrosive | 19% |
| Harmful ^a | 7% |
| Flammable | 75% |
| Oxidizing | 6% |
| Explosive | 30% |

^aSome of the explanations presented for what the “harmful” (St. Andrew’s Cross) symbol meant were: forbidden, no parking, call the Red Cross, do not open, Nazi, out of use, expired, sliding, medicine, take care of your hair, zebra crossing, no smoking, not too serious, and road under construction.

Source: Ref.^[9].

International Labor Office (ILO) recommendations^[7,8] emphasize the need for label understandability. However, what is meant by “understandable” is not defined in detail. Evidence from developing countries^[2,5,6,9,10] shows that working populations may not understand the technical content of intended safety messages, even though the message appears unambiguous to technical staff. For example, Ethiopian workers showed very low levels of comprehensibility of commonly used pictograms on pesticide labels (see Table 1).^[9]

RISK PERCEPTIONS AND THE TARGET AUDIENCE

Perception of risk is highly sensitive to cultural differences and notions of risk are, at least in part, socially constructed. For example, research suggests that the ease of imagining a hazard determines the subjective risk perceived.^[1,2,5] Non-western subjects may have difficulty in imagining risks framed in biomedical models of disease when they hold traditional views of illness and disease, couched in differing cultural beliefs. Similarly, the notion that the severity of the injury is a greater determinant of risk perception for an individual than the likelihood of such injury has not been tested among nonwestern populations.

An assumption of professionals designing pesticide labels is that laypersons share their frame of reference and perceive pesticide risks similarly. However, if the message is not appropriately matched to the frame of reference of the target audience, then communication may fail (or even prove counterproductive). For example, triple rinsing of pesticide containers, routinely advocated by the industry as a safety precaution before destruction, did not deter rural South African women from believing that containers can be “rinsed” for reuse with a cattle dung or soap solution.^[10]

GLOBAL HARMONIZATION OF PESTICIDE LABELING

In 1990, the ILO initiated a project to harmonize existing systems for the classification and labeling of all chemicals, including pesticides. Central to the task of harmonization was the need for a “globally harmonized hazard classification and compatible labeling system, including SDS and easily understandable symbols.”^[2] This goal was endorsed by the Interorganization Program for the Sound Management of Chemicals (IOMC), which has overseen the development of a Globally Harmonized System for the Classification and Labeling of Chemicals (GHS) (Fig. 3). In 1998, the ILO established a tripartite Working Group (WG) for the Harmonization of Chemical Hazard




| GHS Symbol | Indicates |
|---|----------------------|
|  | Very toxic; toxic |
|  | Acute health hazard |
|  | Environmental hazard |

Fig. 3 Selected GHS hazard communication symbols for pesticide labels. (From UN ECOSOC Subcommittee of Experts on the GHS Allocation of Label Elements, ST/SG/AC.10/C.4/2001/25, Geneva, October, 26, 2001.)

Communication, whose brief included developing hazard communication tools to convey information about the harmonized classification criteria. With completion of the GHS, a United Nations Subcommittee on the GHS was established in 1999 to maintain and update the standard. The subcommittee met for the first time in 2001. Formal publication of the GHS document is expected in 2003.

EVALUATING HAZARD COMMUNICATION

Because the vast majority of research studies have been conducted among consumers, workers, and volunteer populations (often students) in developed countries, the effectiveness of hazard labeling for developing country populations is largely unknown. Empirical evaluation of hazard communication is essential to assess whether the intended message is actually reaching target audiences and should ideally incorporate:

1. "Real" world product use scenarios with adequate contextual cues.
2. Cultural specificities in the design and implementation of testing.
3. Tests that measure comprehension rather than test familiarity.
4. Sampling strategies to select subjects that take account of workplace power relations and consumer autonomy.

Evaluations should address access (Do target groups have access to labels?), content (Do they understand?), and impact (Are health and safety improved?).

CONCLUSION

Although global harmonization may play an important role in standardizing classifications, establishing minimum criteria for communicating safety information, and facilitating regulatory enforcement of pesticide labeling, it will not address local specificity in end-user comprehension of hazard communication. For that reason, culturally sensitive methods to develop and evaluate labels and SDS remain critical. Hazard communication evaluations should inform local, national, regional, and global training and education initiatives, and shape national and global policy. Although true for all countries, this need is particularly urgent in developing countries, where the burden of pesticide exposure on human health and the environment is heaviest, and where resources to manage pesticides safely are least available.

See also Ethical Aspects of Pesticide Use; Federal Insecticide, Fungicide, and Rodenticide Act; Pesticide Label Regulations; Regulating Pesticides; Economic and Social Aspects; Decision Making; Legal Aspects of Pest Management and Pesticides; Legal Aspects of Pesticide Application; Pictograms; Product Stewardship and Responsible Care; Safe Use: A Developing Country's Point of View; Safe Use: The Industry's Point of View; and Worker Protection Standard.

ACKNOWLEDGMENT

The authors thank the ILO for permission to reproduce materials used to develop its Hazard Communication Comprehensibility Testing Manual, Mr. Isaac Obadia of the ILO for his comments, and the IOMC Harmonization Working Group for their feedback.

REFERENCES

1. Weyman, A.K.; Kelly, C.J.; Sreenivasan, B. *Overview of Research Findings on Hazard Warning Labels. Supplement to Main Review of the Literature on Risk Perception and Risk Communication in the Workplace (Draft)*; Health and Safety Laboratory, Health and Safety Executive: London, 1998.
2. London, L.; Myers, J.E.; Dalvie, M.A.; Rother, H.A.; Maruping, M.; Miller, S. *Hazard Communication Comprehensibility Study*; Report to the International Labour Office; International Labour Office: Geneva, 2000.
3. Dollimore, L.S. Safer packaging and labeling of pesticides. In *Impact of Pesticide Use on Health in Developing Countries*; Proceedings of a Symposium Held in Ottawa, Canada, September 17–20, 1990; Forget, G.,

- Goodman, T., de Villiers, A., Eds.; International Development Research Center: Ottawa, 1993; 158–165.
4. Sattler, B.; Lippy, P.H.B.; Jordan, T.G. *Hazard Communication: A Review of the Science Underpinning the Art of Communication for Health and Safety*; International Labour Office: Geneva, 1997. Second Draft Document Submitted to ToxaChemica International, Subcontract to the Occupational Health and Safety Administration. Environmental Health Education Centre, University of Maryland Medical School. Documentation for the Working Group on Harmonising Chemical Hazard Communication. Document IOMC/ILO/HC2/99.Inf.2.
 5. Baloyi, R.S. *Report, Occupational Health and Safety Branch*; International Labour Office: Harare, 1997.
 6. Clevestine, E. *Comprehension of Pictograms and Data Sheet Phrases: An African Survey*; International Labour Office: Geneva, 1994.
 7. International Labour Organisation. *Recommendation Concerning Safety in the Use of Chemicals. Recommendation 177*. International Labour Office: Geneva, 1990.
 8. International Labour Organisation. *Safety and Health in the Use of Agrochemicals: A Guide*. International Labour Office: Geneva, 1991.
 9. Berhanu, B. Assessment of workers' knowledge of chemical safety symbols—case study in Ethiopia. *Afr. Newsl. Occup. Health Saf.* **1993**, *3*, 22–23.
 10. Rother, H.A. Influences of pesticide risk perception on the health of rural South African women and children. *Afr. Newsl. Occup. Health Saf.* **2000**, *10* (2), 42–46.

Health Impacts in Developing Countries

Aiwerasia V.F. Ngowi

Pesticide Environmental Management Centre, Tropical Pesticides Research Institute, Arusha, Arusha, Tanzania

Catharina Wesseling

Central American Institute for Studies on Toxic Substances (IRET), Universidad Nacional, Heredia, Costa Rica

Leslie London

Occupational and Environmental Health Research Unit, School of Public Health and Primary Health Care, University of Cape Town, Observatory, Western Cape, South Africa

INTRODUCTION

The use of potentially hazardous chemicals is increasing in developing countries whose populations have the least capacity to protect themselves. Hundreds of thousands of people die annually from the effects of use, misuse, or accidental exposures to pesticides.^[1] Developing nations in Africa, Asia, and Latin America comprise more than 75% of the total world population, use 25% of the world's pesticides, yet account for 99% of deaths caused by these toxins.^[2]

HEALTH IMPACTS

Acute Pesticide Poisoning

More than a decade ago, the World Health Organization estimated that three million cases of acute pesticide poisoning resulting in 220,000 deaths occur worldwide each year, the majority in developing countries.^[2] However, it is well recognized that these figures are an underestimate because of underdiagnosis and/or underreporting. Diagnostic difficulties are prominent in developing countries,^[3,4] owing to insufficient medical training and high background levels of ill health.

Organophosphorus insecticides are the most common agents involved in acute pesticide poisonings, accounting for between 50% and 80% of all poisonings in Asia^[5] and are a major public health concern in most African countries, where approximately 80% of the workforce is in agriculture. In Central America, organophosphates, carbamates, and paraquat account for over 80% of poisonings.

Part of the reason for this picture is the continued use in developing countries of pesticides no longer registered for use in the developed world, because of their high toxicity and the substitution of persistent

organochlorines with organophosphate insecticides. Fatality rates and lifelong disability resulting from pesticide poisoning in developing countries are exacerbated by poor diagnosis and delayed treatment, resulting in both human suffering and economic losses.

High rates of unintentional poisoning, mostly occupational, have been reported in rural agricultural working populations worldwide.^[6,7] Mass poisonings by pesticides in developing countries have typically resulted in high numbers of fatalities. An epidemic of Malathion poisoning in Pakistan, in 1976, resulted in five deaths and approximately 2800 acute poisoning episodes.^[8] In the remote Andean village of Taucamarca in October 1999, 42 children were poisoned after eating a school breakfast contaminated with the organophosphate pesticide methyl parathion, resulting in 24 deaths before the children could reach medical treatment.^[9]

However, it is only a limited number of the most extreme cases in developing countries, which appear to be documented. Less high-profile cases are common but unrecorded. For example, a methomyl-poisoning incident involving 11 female flower farm workers in Arusha, Tanzania in March 2004 was reported in the press, but absence of adequate local investigation mechanisms prevented its documentation in the peer-reviewed literature.

Deliberate self-harm is a major problem in the developing world. Pesticides are commonly used as agents throughout developing nations, and are associated with high mortality rates.^[1,6,7] In Surinam, the incidence of suicide with paraquat correlated closely with amounts of paraquat imported and used in agriculture. In India, suicide using aluminum phosphide was reported as so common that postmortem examinations on deceased bodies were said to be routinely conducted by staff wearing respirators for personal protection.^[10,11]

Underlying factors that make individuals at risk for self-harm are both social (including domestic problems,

poverty, social isolation, and financial hardship) and medical.^[1] Farmer indebtedness, widespread in many developing countries characterized by unequal economic systems, is an important factor driving high rates of suicide. More recent findings suggest that pesticides, particularly organophosphates, may be more than agents in suicidal attempts, but also part of the causal pathway because of their neurotoxicity and the possible links between organophosphate exposure, depression, and impulsivity, mediated through effects on neurotransmitters such as serotonin.^[6] In a context where the above social risk factors for depression are common in developing countries, further exposure to neurotoxic pesticides may substantially increase the risks of suicide.

Chronic Health Impacts Unknown

Although long-term consequences of pesticide poisoning are well recognized in the literature, relatively few studies of long-term health effects of pesticide exposure have been conducted amongst working populations in developing countries. Underdiagnosis is accentuated for long-term health consequences that require greater diagnostic capacity. Dermal exposure routes for developing country workers are also a common but underdocumented yet critical pathway for systemic poisonings, both acute and chronic. Consequently, the extent of chronic health impacts of pesticides in developing country workers is poorly characterized. However, there is little reason to believe their impact would be any less than in developed countries. Indeed, high levels of background morbidity and poor social conditions are likely to aggravate pesticide toxicity. For example, research amongst South African farm workers highlighted the link between chronic lifetime undernutrition, organophosphate exposure, and impaired neurological performance on tests of vibration threshold.^[12] Azoospermia, oligospermia, and low fertility have been documented in over 26,000 workers, previously exposed to 1,2-dibromo-3-chloropropane (DBCP) on banana and pineapple plantations in over 12 countries.^[13]

WEAK SURVEILLANCE FOR HAZARDS AND IMPACT

Although a critical public health tool for the control of pesticide poisoning, surveillance in developing countries is bedeviled by multiple problems such as lack of access to health care for poisoning survivors, lack of human resources, diagnostic skills and equipment to identify cases, and weak information systems. Acute poisoning rates are consequently underestimated and may selectively undercount certain types of poisoning (occupational circumstances) and certain risk groups

(women and migrant workers). Lack of professional competence and conflict of interest arising from compensation system levies may also lead occupational poisonings to be misreported as suicide.^[7] As a result, inferences from review of flawed data may lead to mistaken policy decisions.^[14]

To improve information on the extent of pesticide poisoning in developing countries, surveillance systems for acute health effects from pesticides are being established in developing nations. In 1998, almost 6000 pesticide poisonings were reported in five of the seven Central American countries generating an estimate, corrected for underreporting, of 30,000 pesticide poisonings annually in the region.^[9] Poisoning rates reported in an intensified surveillance intervention in South Africa increased 10-fold in the study area compared to a control area.^[14] Similarly, the International Program on Chemical Safety has initiated the piloting of a surveillance tool to derive better estimates of the extent of global pesticide poisoning.^[15]

WEAK REGULATION AND ENFORCEMENT

Vulnerable economies and weak infrastructure in developing nations hinder their ability to regulate the use of pesticides, particularly when macroeconomic pressures promote deregulation and restrict public spending required to implement regulatory controls. As a result, marketing and advertising of pesticides are often uncontrolled. Incorrectly labelled or unlabelled formulations, including ready-made solutions in soft drink bottles and other containers, are commonly sold at open stands. In South Africa, the repackaging of aldicarb granules into small-volume packets sold by street vendors for domestic pest control has been linked to increasing numbers of suicides in urban areas. Low retail prices, sometimes associated with subsidy policies, promote risky pesticide use. Weaknesses in sustainable international and national agricultural and chemicals management policies manifest in a reliance on “safe-use” strategies. Yet, evidence has shown that the so-called “good agricultural practices” and “safe use” are ineffective in controlling risks in developing countries, principally because many measures assumed to enable safe use are not feasible in developing countries, particularly under tropical or adverse climatic conditions.

LOW LEVELS OF WORKER AND COMMUNITY AWARENESS

Farmers and farm workers rarely have access to adequate training in pesticide safety or advice on the complicated management of pesticides. Hot climates are a disincentive to use of protective clothing, and

many workers and farmers lack access to water for washing hands or exposed skin, increasing the risks of contamination. Recognition of pests and their predators is generally low, leading to overreliance on routine pesticide applications to control pests; knowledge of product selection, application rates, and timing is poor; different products are often combined in the belief that the effect will be greater; reentry periods after spraying are not known; and without knowledge of alternatives, farmers often assume that the only solution to pest problems is to spray more frequently.^[9] Pesticides are often stored improperly in or around farmers' homes, increasing family members access.^[16] In some instances, empty pesticide containers are reused to store water and food, resulting in serious poisonings.

IMPORT/EXPORT OF BANNED AND RESTRICTED COMPOUNDS

Pesticides banned or restricted in developed countries are often easily available in developing countries. These include pesticides causing significant acute and chronic morbidity (such as class I and II organophosphates and paraquat) and organochlorines earmarked for eradication under the Stockholm Convention on persistent organic pollutants (POPs) (particularly dieldrin, lindane, and chlordane). Endosulfan, a candidate pesticide for inclusion under the POPs treaty, has been responsible for a series of poisonings in Benin^[17] and developmental impacts on children in Kerala, a state in India.^[17,18]

The use of *p-p*-dichlorodiphenyltrichloroethane (DDT) continues to be permitted for malaria control in developing countries, where malaria remains endemic, despite its known hazards for wildlife and controversial adverse effects on human health. As a result, it is still produced for export in at least three countries. Because of its ongoing usage for public health vector control, unauthorized use for agricultural purposes remains a concern in developing countries, particularly where regulatory controls are weak. The presence and persistence of DDT and its metabolites worldwide are still problems of great global relevance to public health.

LACK OF TECHNICAL AND LABORATORY CAPACITY

Many developing countries suffer from a lack of human and technical resources, aggravated by the global brain drain and weak economies. As a result, few developing countries are able to monitor pesticide residues. Most countries do not have laboratories capable of

conducting analyses for pesticides and their residues, particularly at standards that meet good laboratory practice. Where laboratory capacity is available, it is usually to service residue testing of agricultural exports destined for consumers in developed countries. Produce grown for domestic consumption is rarely monitored.

Environmental media such as water and soil are rarely tested, and, even then, usually only on a research basis. Isolated studies of lactating women in Southern Africa have confirmed the presence of high levels of DDT metabolites in breast milk in populations living in malaria endemic areas subject to DDT applications. Yet, despite provisions arising from the POPs treaty to undertake routine testing to monitor the impact of DDT use, there is no system for biological monitoring for DDT metabolites in place in Southern Africa. As a result, many infants in the region are substantially exposed through cross-placental transfer and breastfeeding, with potential adverse impacts on childhood neurodevelopment.

Research capacity to identify problems and develop prevention strategies is also constrained by limited investments in capacity building in relevant scientific fields. As a result, there is neither proactive monitoring, nor the use of information systems to effect adequate responses to pesticide problems identified.

PEST CONTROL POLICIES

Unlike many developed countries, agricultural policies in many developing countries have emphasized short-term economic gains at the expense of environmental sustainability or human health. Few developing countries have adopted integrated pest management or pest reduction strategies. The dominant "pesticide culture" assumes that the use of pesticides to control pest as the first option is the norm, is reinforced by advertising and marketing practices, and is often encouraged by agricultural credit policies and development aid. Much needs to be done to enhance research and development to support pesticide reduction for agriculture and public health, and to strengthen the capacity in developing countries to develop monitoring systems and research capacity to deal with the problems of pesticides in developing nations.

CONCLUSIONS

Underestimations of acute and long-term effects of pesticide in developing countries occur due to under-diagnosis and/or underreporting. The impact of pesticide poisoning is also unknown because of weak surveillance for hazards and impact; import/export of banned or restricted compounds; lack of

technical and laboratory capacity; weak regulations and enforcement; low level of worker and community awareness as well as inappropriate pest control policies. Enhancing research and development to support pesticide reduction for agriculture and public health and strengthening capacity to develop monitoring systems is the best option available for developing countries to deal with the problems of pesticides.

REFERENCES

- Konradsen, F.; van der Hoek, W.; Cole, D.C.; Hutchinson, G.; Daisley, H.; Singh, S.; Eddleston, M. Reducing acute poisoning in developing countries—options for restricting the availability of pesticides. *Toxicology* **2003**, *192* (2–3), 249–261.
- WHO. Public Health Impact of Pesticide Used in Agriculture. Geneva, World Health Organization and United Nations Environment Programme, 1990.
- Mbakaya, C.F.L.; Ohayo-Mitoko, G.J.A.; Ngowi, A.V.F.; Mbabazi, R.; Simwa, J.M.; Maeda, D.N.; Stephens, J.; Hakuza, H. The status of pesticide usage in East Africa. *Afr. J. Health Sci.* **1994**, *1*, 37–41.
- London, L.; Myers, J.E. Critical issues for agrochemical safety in South Africa. *Am. J. Ind. Med.* **1995**, *27*, 1–14.
- He, F.; Xu, H.; Quin, F. Intermediate myasthenia syndrome following acute organophosphates poisoning—an analysis of 21 cases. *Hum. Exp. Toxicol.* **1998**, *17*, 40–45.
- London, L.; Flisher, A.J.; Wesseling, C.; Mergler, D.; Kromhout, H. Suicide and exposure to organophosphate insecticides: cause or effect. *Am. J. Ind. Med.* **2005**, *47*, 308–321.
- Wesseling, C. Multiple health problems in Latin America. In *Silent Invaders: Pesticides, Livelihoods and Women's Health*; Jacobs, M., Dinham, B., Eds.; ZED Books: London, 2003.
- Alderman, L.; Baker, E.L., Jr.; Dobbin, R.D.; Miles, J.W.; Miller, S.; Teeters, W.R.; Warren, M.; Zack, M. Epidemic malathion poisoning in Pakistan malaria workers. *Lancet* **1978**, *1* (8054), 31–34.
- Rosenthal, E. The tragedy of Taucamarca: a human rights perspectiva on the pesticida poisoning deaths of 24 children in the Peruvian Andes. *Int. J. Occ. Env. Health* **2003**, *9*, 53–58.
- Levine, R.S.; Doull, J. Global estimates of acute pesticide morbidity and mortality. *Rev. Environ. Contam. Toxicol.* **1992**, *129*, 29–50.
- Perriens, J.; Van der Stuyft, P.; Chee, H.; Benimadho, S. The epidemiology of paraquat intoxications in Surinam. *Trop. Geogr. Med.* **1989**, *41*, 266–269.
- London, L. Occupational epidemiology in agriculture: a case study in the Southern African context. *Int. J. Environ. Occup. Health* **1998**, *4*, 245–256.
- Slutsky, M.; Levin, J.L.; Levy, B.S. Azoospermia and oligospermia among a large cohort of DBCP applicators in 12 countries. *Int. J. Occup. Environ. Health* **1999**, *5* (2), 116–122.
- London, L.; Bailie, R. Challenges for improving surveillance for pesticide poisoning: policy implications for developing countries. *Int. J. Epidemiol.* **2001**, *30* (3), 564–570.
- Osorio, A.M. Surveillance systems for pesticide intoxications. *Int. J. Occup. Environ. Health* **2002**, *8*, 1–13.
- Ngowi, A.V.F.; Maeda, D.N.; Wesseling, C.; Partanen, T.J.; Sanga, M.P.; Mbise, G. Pesticide handling practices in agriculture in tanzania: observational data on 27 coffee and cotton farms. *Int. J. Occup. Environ. Health* **2001**, *7*, 326–332.
- Dinham, B.; Malik, S. Pesticides and human rights. *Int. J. Occup. Environ. Health* **2003**, *9*, 40–52.
- Saiyed, H.; Dewan, A.; Bhatnagar, V.; Shenoy, U.; Shenoy, R.; Rajmohan, H.; Patel, K.; Kashyap, R.; Kulkarni, P.; Rajan, B.; Lakkad, B. Effect of endosulfan on male reproductive development. *Environ. Health. Perspect.* **2003**, *111*, 1958–1962.

Helicoverpa armigera: Ecology and Control Using Novel Biotechnological Approaches

Vaijayanti A. Tamhane

Ashok P. Giri

Vidya S. Gupta

Plant Molecular Biology Unit, Division of Biochemical Sciences, National Chemical Laboratory, Pune, Maharashtra, India

INTRODUCTION

Helicoverpa armigera (Hübner, Notudae) is a devastating pest that affects many important crop plants. Owing to the intensive agricultural practices, the insect has acquired a pest status. Chemical pesticides are used to control the insect outbreak in the fields, although this routine method of pest control is not a long-term solution. Many biological control methods are coming up, which if applied on a large scale can control the pest and also reduce environmental pollution. Novel biotechnological approaches like use of transgenics with *Bacillus thuringiensis* (Bt) toxin and/or proteinase inhibitor (PI) genes have shown success in insect control. Many Bt transgenic crops are grown all over the world, but being a toxin, may suffer a drawback of developing quick resistance in the insect population. Proteinase inhibitors and other growth retardants like amylase inhibitors and lectins, if combined in transgenics, may play an important role. Efforts need to be taken to keep a balance between the pest and its natural enemies present in the ecosystem. Such a method considers the balance of field ecosystem as the primary way of pest management and the use of any other method of pest control as a supportive one.

BACKGROUND

H. armigera is the most serious polyphagous pest that affects about 181 plant species all over the world. It can complete its life cycle on hosts like cotton, pulses (chickpea, pigeonpea, and sweet pea), maize, tobacco, soybean, rapeseed, groundnut, safflower, sunflower, sorghum, potato, vegetables (tomato, okra, and cauliflower), some forest trees, and fruits. It attacks leaves, tender shoots, apical tips, flower buds, and pods of various crop plants, and can account for up to 90% reduction in yield. The infestation of *H. armigera* causes a worldwide loss of US\$ 7.5 billion despite the use of insecticide worth US\$ 2 billion.^[1] By enhancing insect resistance of these host plants and controlling the insect population, the yields can be increased by at least three times.^[2]

ECOLOGY OF THE PEST

H. armigera is known by various common names like bollworm when it feeds on cotton, pod borer when it feeds on chickpea and pigeonpea, fruit borer when it feeds on tomato, and earworm when it feeds on corn (Figs. 1A and 1B).

It lays yellowish white eggs on lower surface of leaves, flowers, shoot tips, and young pods. Just before hatching, the eggs become dark brown in color. The first instar larvae feed on young leaves. In the later instars, the larvae travel to the pods, bore into it, and feed upon the developing seeds in case of pulses and on boll in case of cotton, thus affecting the agronomic yield of the plant. In the larval stage, *H. armigera* undergoes molting around five or six times, thus giving rise to the six larval instars. A single larva eats up to 8–17 pods in its lifetime. Fully grown larvae drop on the ground and enter the soil up to 2–6 cm below the surface and undergo pupation. The incubation period for the eggs laid is around 3–4 days, larval stage lasts for around 12–16 days, the pupal stage lasts for about 6–10 days in normal condition, and the moth stage lasts for about 6–7 days (Fig. 2).

During winter, the pupae have a 110-day pupal duration, and moths emerge out on the onset of warmer weather, which coincides with the podding stage of the hosts like chickpea and pigeon pea, and therefore a severe attack by the insects is seen. *H. armigera* is well adapted in the habitats created by intensive agriculture and attains a major pest status because of its polyphagous nature, multiple generations, high reproductive rate, and ability to undergo diapause.^[3]

CHEMICAL PESTICIDES

Use of chemical pesticides like chlorinated hydrocarbons, organophosphates, synthetic pyrethroids, and carbamates is preferred by the farmer, as these can bring about a total control of *H. armigera*, which ultimately increases the productivity of the crop. However, owing to their recommended usage, insects have

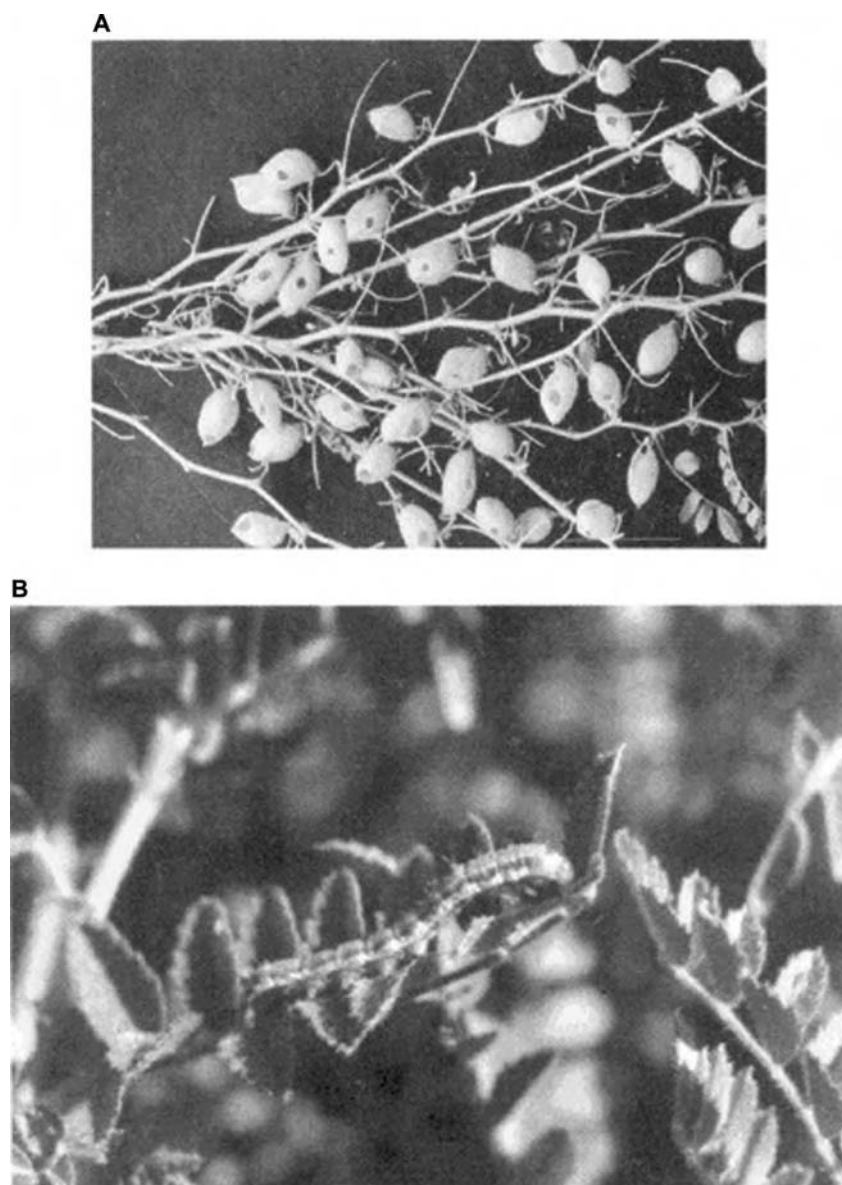


Fig. 1 (A) Damage to the chickpea pods caused by an *H. armigera* attack. Most of the pods are seen with holes bored by the pests. (B) *H. armigera* larva feeding on chickpea plant.

developed resistance to the pesticides. It is recommended to use the pesticides alternately with different modes of action, so that the insects do not develop resistance quickly and the efficacy of control of insects can be maximized with minimum harmful effects. Apart from this, the chemical pesticides have a drawback of being detrimental to the ecosystem by attacking and wiping out the target and the nontarget species, thus eradicating the natural enemies of the pest. They cause pollution of land, water, and air, and enter into the food chain of higher animals. In birds, they affect the reproductive system, and therefore are shown to be a threat to the existence of several wild organisms. Accumulation of pesticides in human beings is reported to cause serious health hazards like cancer.

NATURAL SYSTEMS/AGRICULTURAL PRACTICES

Reducing the extent of crop damage by the pest is the aim of most of the agricultural practices. The farmers carry out these methods by manipulating the time of sowing, cropping season, spacing, and fertilizer application. Deep plowing and intercultural operations reduce the survival and build up of *H. armigera* population. Hand picking the large sized larvae, shaking the plants to make the larvae fall down, which are later picked up and destroyed are some of the management methods.^[4]

H. armigera is a protein-rich food for many bird predators. Having smaller fields with lining of hedge plants for resting of birds is advisable to get a natural

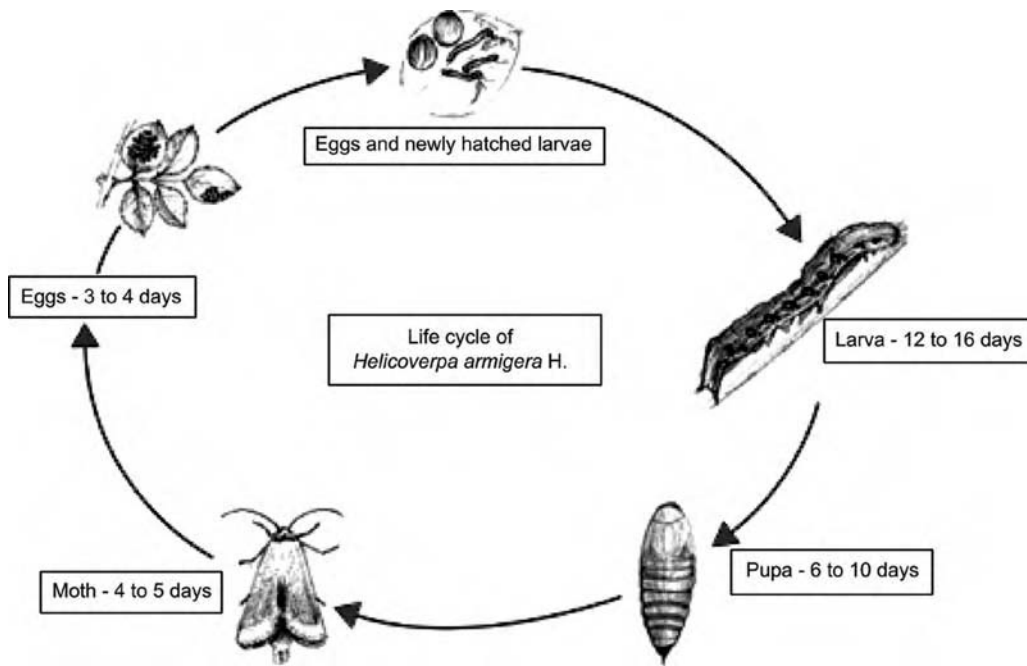


Fig. 2 Life cycle of *H. armigera*.

control over the insect population. However, owing to intensive agriculture, these practices are becoming rare.

Neem (*Azadirachata indica*) oil 1% and kernel oil 5% can also be sprayed but with moderate results.^[2] However, this needs to be further explored for their universal use and unlimited availability.

BIOLOGICAL CONTROL METHODS

Pheromones

Pheromones are sex hormones emitted by the female moth for attracting the males. Glands producing the chemical attractant are located in the lower segment of the abdomen of the female moth. Using pheromones is a species-specific, non-toxic, and environment-friendly method of insect control. The chemical signal responsible for the attraction of the male moths is identified and applied in a trap, which is then installed in the field. The trap releases the chemical signal in the air and moths are attracted to and trapped in it. This method thus reduces the moth population in the next generation.

H. armigera Nuclear Polyhedrosis Virus

Use of *H. armigera* nuclear polyhedrosis virus (HaNPV), a viral pathogen that is specific for *H. armigera*, has shown promising results to control the pest.^[5] This

virus enters the insect gut along with the food. It penetrates into the epithelial cells, multiplies rapidly, and spreads throughout the insect body, finally killing the insect. Although effective, NPV sprays are not readily available, have maintenance and application drawbacks, and are not cost-effective.

Use of *Trichogramma* sp., which is an egg parasitoid, and entamopathogenic fungi *Beauveria bassiana* and *Metarrhizium anisopliae*, and *Pseudomonas*, has been found to be moderately effective. Insect parasites like *Campoletis chloridae*, *Apanteles ruficrus*, and *Carcelia illota* can infect the larval stage of the pest and can be used for its control. *Araneus nympha* and *Oxyopes shewta* are predatory spiders, which attack the larval stage of the pest.

B. thuringiensis Sprays

B. thuringiensis is a gram-positive bacterium, which on sporulation produces a protoxic protein. This protein, which is in the form of a proprotein, gets converted into a toxin by activity of the proteinases in the gut, when ingested by the insect. The active Bt toxin permeates the gut epithelial membrane, thus impairing the digestion and reducing its feeding capacity. The toxin also spreads to other tissues of the insect body and accumulates to finally kill the insect pest. The toxin has little or no effect on plants, mammals, and predatory insects. The spores of Bt in the form of a suspension are used to spray in the *H. armigera*

infested field. The larvae eat up the spores and thus get killed.

All these biocontrol methods, though environment-friendly and target-specific, are not widely used, primarily because of their high production costs and sparse availability. However, when consumption of insecticides has grown beyond proportions, there is an urgent need to refine these techniques to make them as effective as the chemical pesticides and to make them commercially viable.

BIOTECHNOLOGICAL APPROACHES

Because of the lack of resistance sources in the primary and secondary gene pools of the hosts of *H. armigera*, including chickpea, pigeonpea, cotton, etc., conventional breeding methods are of little use to develop *H. armigera* resistance in host plants.

Transgenics with Bt Endotoxin

B. thuringiensis toxin gene has been identified, isolated, and characterized, and has been used for the development of transgenic chickpea. *H. armigera* is sensitive to a range of Bt toxins, CryIAC being the most effective one. Combination of Bt toxins like CryIAC and CryIf is found to enhance resistance. Commercialization of Bt transgenic cotton has been carried out and other crops have also been successfully transformed with these Bt toxin genes. It is observed in cotton that *H. armigera* populations from different geographic areas show fivefold variation in their sensitivity to CryIAC transgenics. In chickpea, two cultivars ICCV1 and ICCV6 have been transformed to express the CryIAC gene under a constitutive promoter (CaMV35s). Young shoots of T1 plants express CryIAC protein at 0.0045% of soluble protein, which causes feeding inhibition on the first instar larvae.^[1] Transgenics expressing Bt toxin, though very effective in *H. armigera* control,^[6,7] is only a short-term approach as the insect soon develops resistance to the Bt toxin and the transgenics may become ineffective in few generations owing to the lethal effect of Bt on the plants.

A new protein VIP has been isolated from *B. thuringiensis* and is shown to be effective against *H. armigera*.

Transgenics with Plant Proteinase Inhibitors

H. armigera possesses an array of proteinases in its gut for digestion of ingested food, predominantly serine proteinases like trypsin, chymotrypsin, and elastase, and other proteinases such as aspartic and metallo proteinases and amino- and carboxy exopeptidases. It has

been reported that the *H. armigera* gut proteinase composition changes according to the host plant that it feeds on.^[8,9]

Proteinase inhibitors are ubiquitously found in the plant kingdom and have been involved in defense. Serine PIs inhibit the proteinases from the insect gut, and thereby create a stress on the digestive system of the pest. Improper digestion of the ingested food leads to scarcity of amino acids and thus results in growth retardation. The insect has to invest energy for synthesizing more or different types of the proteinases to overcome the inhibition. The inhibitor fed insects have also shown severely reduced fecundity and fertility, thereby affecting the exponential rise in the population. This approach of insect control does not wipe out the insect population completely, and thereby prevents development of resistance in the insect population. On ingestion of PIs, the insect is under stress and gets exposed to other conditions of attack by the predators.

The PI genes from several plants have been transferred to crop plants and the transgenics have revealed insect resistance at a laboratory scale or in greenhouse trials. The *H. armigera* proteinases can inactivate or digest the host PIs and use them as amino acid pool. The PIs from certain non-host plants (e.g., winged bean, bitter melon, and hot pepper), to which the pest has never been exposed to, are suitable for inhibiting insect gut proteinases. Proteinase inhibitor genes from these plants have been isolated, and attempts are underway to transfer them to chickpea. Similar to PIs, lectins and amylase inhibitors have also been characterized and can be employed for developing transgenics that are tolerant to insect pests.

Problems with plant PI strategy and possible solutions

Two major constraints of PI-based insect control strategy are that the insects adapt themselves to the expressed PI protein and PI expression under constitutive promoter might reduce the plant fitness.^[10] The PI might interfere with the metabolic process of the plant. Tissue-specific expression of the PI under wound-induced or insect feeding-induced promoter can target the expression of the PI on insect attack only and reduce the undesirable interference of PI in the plant's metabolic processes.

CONCLUSIONS

It is very clear that the pest-management approach should safeguard ecological sustainability.^[11] Maintaining a natural equilibrium of pests and predators

in the field combined with applying novel biotechnological approaches for the host crop tolerance keeps the pest in acceptable bounds. It is therefore essential to emphasize on the following aspects:

1. Understanding the insect response to the ingested biomolecules such as PI, AI, lectin, etc.
2. Analysis of the effect of ingested biomolecules PI(s) on *H. armigera* population.
3. Identification of appropriate biomolecule combinations and testing their efficacy.
4. Analysis of agronomic behavior of the transgenic crops that express biomolecules.

REFERENCES

1. Gowda, C.L.L.; Sharma, H.C. Legume pod borer/cotton bollworm, *Heliiothis/Helicoverpa*—the global problem. In *Souvenir National Symposium on Helicoverpa Management*; National Challenge, Kanpur, India; Panwar, J.D.S., Ahlawat, I.P.S., Lal, S.S. Eds.; Indian Institute of Pulse Research: Kanpur, 2005; 1–9.
2. Romeis, J.; Sharma, H.C.; Sharma, K.K.; Das, S.; Sharma, B.K. The potential of transgenic chickpeas for pest control and possible effects on non-target arthropods. *Crop Prot.* **2004**, *23*, 923–938.
3. Ahmad, R.; Rai, A.B. Biology and ecology. In *25 Years of Research on Helicoverpa at IIPR*; Upadhyaya, D., Ali, M., Eds.; Indian Institute of Pulse Research: Kanpur, Lucknow, 2005; 7–16.
4. Shanower, T.G.; Kelley, T.G.; Cowgill, S.E. Development of effective and environmentally sound strategies to control *Helicoverpa armigera* in pigeonpea and chickpea production systems. Proceedings of the Third International Conference on Tropical Entomology, Saini, R.K., Ed.; ICIP Science Press: Nairobi, Kenya, 1998; 239–260.
5. Cherry, A.J.; Rabindra, R.J.; Parnell, M.A.; Geetha, N.; Kennedy, J.S.; Grzywacz, D. Field evaluation of *Helicoverpa armigera* nucleopolyhedrovirus formulations for control of the chickpea pod-borer, *H. armigera* (Hubn.), on chickpea (*Cicer arietinum* var. Shoba) in southern India. *Crop Prot.* **2000**, *19*, 51–60.
6. Wu, K.; Guo, Y.; Lv, N.; Greenplate, G.T.; Deaton, R. Resistance monitoring of *Helicoverpa armigera* to *Bacillus thuringiensis* insecticidal protein in China. *J. Econ. Entomol.* **2002**, *95*, 826–831.
7. Wilkins, T.A.; Rajasekaran, K.; Anderson, D.M. Cotton biotechnology. *Crit. Rev. Plant Sci.* **2000**, *19*, 511–550.
8. Patankar, A.G.; Giri, A.P.; Harsulkar, A.M.; Sainani, M.N.; Deshpande, V.V.; Ranjekar, P.K.; Gupta, V.S. Complexity in specificities and expression of *Helicoverpa armigera* gut proteases explains polyphagous nature of the insect pest. *Insect Biochem. Mol. Biol.* **2001**, *31*, 453–464.
9. Chougule, N.P.; Giri, A.P.; Sainani, M.N.; Gupta, V.S. Gene expression patterns of *Helicoverpa armigera* gut proteases. *Insect Biochem. Mol. Biol.* **2005**, *35*, 355–367.
10. Zavala, J.A.; Patankar, A.G.; Gase, K.; Baldwin, I.T. Constitutive and inducible trypsin proteinase inhibitor production incurs large fitness costs in *Nicotiana attenuata*. *Proc. Natl. Acad. Sci. U.S.A.* **2004**, *101*, 1607–1612.
11. Lewis, W.J.; Lenteren, J.C.V.; Pathak, S.C.; Tumlinson, J.H., III. A total system approach to sustainable pest management. *Proc. Natl. Acad. Sci. U.S.A.* **1997**, *94*, 12,243–12,248.

Hormonal Disruption in Humans

Evamarie Straube

*Institute of Occupational Medicine, University of Greifswald,
Greifswald, Germany*

Sebastian Straube

*Department of Physiology, Laboratory of Molecular and Cellular Signalling,
University of Oxford, Oxford, U.K.*

Wolfgang Straube

*Department of Gynecology and Obstetrics, University of Greifswald,
Greifswald, Germany*

INTRODUCTION

Hormonal disruption in humans can result from genetic disorders,^[1] disease,^[2] mental and physical^[3] stress, and, importantly, from chemical exposure, including exposure via nutrition. Substances relevant for chemical exposure include pesticides (organochlorines such as DDT,^[4] other organohalogens such as dibromochloropropane,^[5] some organophosphates, carbamates, dithiocarbamates, phthalates), polychlorinated biphenyls,^[6] some solvents,^[7] metals such as cadmium, lead, and manganese,^[8] phytoestrogens and isoflavonoids.^[9] Furthermore, hormonal disruption can be caused by lifestyle factors such as smoking and alcohol use,^[3] and by certain drugs, e.g., glucocorticoids, hypnotics, anti-hypertensives, neuroleptics, and H₂-antihistaminics.^[2]

Hormonal disruption can affect all endocrine systems. Thyroid hormone inhibition has been reported in humans after occupational exposure to amitrol and mancozeb.^[10] Insulin levels are affected by streptozotocin, which is toxic to pancreatic beta cells.^[11] However, we know most about chemical exposure affecting the reproductive system. Chemicals acting as xenoestrogens (mimicking the action of endogenous hormones) or otherwise interfering with endocrine processes are collectively called endocrine disruptors. An endocrine disruptor chemical (EDC) has been defined as an “exogenous substance that causes adverse health effects in an intact organism, or its progeny subsequent to changes in endocrine functions.”^[12] A list of substances adversely affecting human health, including via effects on reproductive function, has been compiled by the European Community.^[13] Pesticides are perhaps the most important EDCs. Exposure to pesticides has been linked with adverse health effects, ranging from reproductive problems to cancer.^[14]

Guidelines for the use of pesticides exist with regard to general exposure (acceptable daily intake—ADI, tolerable daily intake—TDI) and professional exposure

(maximal allowable concentration—MAC, daily tolerable dermal exposure—Dtol). However, various pesticides are known or suspected to interfere with hormone function even at very low concentrations. At the request of the United States Environmental Protection Agency, “low-dose effects” of some well-known EDCs—bisphenol A, diethylstilbestrol (DES), ethinylestradiol, nonylphenol, octylphenol, genistein, methoxychlor, 17 β -estradiol, and vinclozolin—were evaluated by a peer-review panel.^[15] It was concluded that biological changes could be caused by EDCs in the range of typical human exposure. Some EDCs have non-monotonic dose–response relationships, with hormonal disruption occurring at relatively low levels of exposure.^[16]

MECHANISMS OF CHEMICAL DISRUPTION

Chemical disruption can be caused by xenoestrogens. Xenoestrogens, such as endosulfan, toxaphen, dieldrin, o,p’DDT, bisphenol A, nonylphenols, and dibutylphthalates,^[7] mimic the physiological effects of estrogens. Xenoantiestrogens have effects opposite to those of xenoestrogens. For example, dioxin exerts its inhibitory effect by enhancing the expression of enzymes that degrade the estrogen receptors.^[17] Antiandrogenic effects may result from competitive antagonism at androgen receptors. This was demonstrated for vinclozolin and DDE, the stable metabolite of the DDT.^[7] It is sometimes the case that a xenobiotic and its metabolite (such as DDT and DDE) can exert their effects at different targets in the organism.

Pesticide-induced enzymes such as UDP-glucuronyl transferase and monooxygenases can degrade hormones (e.g., testosterone). Furthermore, the pesticides endosulfan, mirex, and DDT can increase the elimination of androgens by stimulating cytochrome

P-450.^[18,19] Pesticide exposure can also disrupt hormonal status by inhibiting enzymes. For example, inhibition of the aromatase system leads to an increase in testosterone levels and a decrease in the formation of estradiol from testosterone.^[20] Inhibitors of the aromatase system include prochloraz, imazalil, propiconazole, fenarimol, triadimenol, triadimefon, and dicofol.^[21]

HORMONAL DISRUPTION IN WOMEN

It is often difficult to establish a relationship between EDCs and adverse health effects in women. Professional exposure usually affects men, not women. In addition, oral contraceptives or postmenopausal hormone replacement therapy often influences hormonal status to a much larger extent than EDCs on a weight-to-weight basis.

In pregnant women, EDCs may cause miscarriage or malformation, possibly by reinforcing genetic predispositions to disease.^[22] The type of damage typically depends on how far the pregnancy has progressed at the time of exposure. Historically, diethylstilbestrol (DES) was the first recognized example of a xenobiotic eliciting a hormonal effect. In pregnant women, treatment with DES leads to an increase in the incidence of adenocarcinoma of the vagina in their daughters^[23] and malformations of the external genitals in their sons^[24] and grandsons.^[25] Treatment with DES has, furthermore, been reported to have an effect on sexual orientation^[26] and handedness.^[27]

HORMONAL DISRUPTION IN MEN

We have found changes in sex hormone concentrations and T-lymphocyte counts after acute and chronic low-dose professional exposure to pesticides. There were two opposite effects depending on the duration of exposure: a hormonal (similar to the results of Garry et al.^[28]) and immune suppression after acute exposure and an activation of both systems following chronic exposure.^[20] We found a reduction in estradiol levels during and after the application season in pesticide applicators. Of the various classes of pesticides used, organophosphates and carbamates were the most effective hormonal disruptors, presumably acting by inhibiting the aromatase system.^[29] Another study^[22] has found an increase in estradiol concentration in pesticide-exposed men, but, as the timing of the sampling with regard to exposure was not well defined, this might have been due to a rebound effect.

PESTICIDES TEST MANAGEMENT

The Office of Prevention, Pesticides, and Toxic Substances has developed guidelines for the testing of pesticides and toxic substances with regard to effects on reproduction and fertility.^[30] Similar guidelines were developed by the European Commission.^[13] However, both testing paradigms do not, in our opinion, give adequate consideration to low-dose ranges of EDCs, which are associated with hormonal disruption, and do not offer a way of directly determining xenohormone activity (estrogenicity or androgenicity) nor for measuring the inhibition of key enzymes, e.g., aromatase.

A determination of sex hormones (LH, FSH, prolactin, testosterone, and, especially in men, estradiol) should be included in a medical check-up of persons professionally exposed to pesticides.

REFERENCES

1. Achermann, J.C.; Ozisik, G.; Meeks, J.J.; Jameson, J.L. Genetic causes of human reproductive disease. *J. Clin. Endocrinol. Metab.* **2002**, *87* (6), 2447–2454.
2. Turner, H.E.; Wass, J.A.H. Gonadal function in men with chronic illness. *Clin. Endocrinol.* **1997**, *47* (4), 379–403.
3. Vermeulen, A.; Kaufman, J.M. The age associated decline in testicular function: partial androgen deficiency of the ageing male (PADAM). *Menopause Rev.* **1999**, *IV* (2), 23–35.
4. Welch, R.M.; Levin, W.; Conney, A.H. Estrogenic action of DDT and its analogs. *Toxicol. Appl. Pharmacol.* **1969**, *14* (2), 358–367.
5. Potashnik, G.; Porath, A. Dibromochloropropane (DCBP): A 17-year reassessment of testicular function and reproductive performance. *J. Occup. Environ. Med.* **1995**, *37* (11), 1287–1292.
6. Bitman, J.; Cecil, H.J. Estrogenic activity of DDT analogs and polychlorinated biphenyls. *J. Agric. Food Chem.* **1970**, *18* (6), 1108–1112.
7. Massaad, C.; Entezami, F.; Massade, L.; Benahmed, M.; Olivennes, F.; Barouki, R.; Hamamah, S. How can chemical compounds alter human fertility? *Eur. J. Obstet., Gynecol., Reprod. Biol.* **2002**, *100* (2), 127–137.
8. Gennart, J.P.; Buchet, J.P.; Roeis, H.; Ghyselen, P.; Ceulemans, E.; Lauwerys, R. Fertility of male workers exposed to cadmium, lead or manganese. *Am. J. Epidemiol.* **1992**, *135* (11), 1208–1219.
9. Jacobs, M.N.; Lewis, D.F. Steroid hormone receptors and dietary ligands: a selected review. *Proc. Nutr. Soc.* **2002**, *61* (1), 105–122.
10. Cocco, P. On the rumors about the silent spring. Review of the scientific evidence linking occupational and environmental pesticide exposure to endocrine disrupting health effects. *Cad. Saude Publica* **2002**, *18* (2), 379–402.
11. Bolzan, A.D.; Bianchi, M.S. Genotoxicity of streptozotocin. *Mutat. Res.* **2002**, *512* (2–3), 121–134.

12. Harrison, P.T.C.; Holmes, P.; Humfrey, C.D.N. Reproductive health in humans and wildlife: are adverse trends associated with environmental chemical exposure? *Sci. Total Environ.* **1997**, *205* (2–3), 97–106.
13. http://ecb.jrc.it/Documents/Classification-Labeling/28th_ATP.pdf (accessed March 2003).
14. Skakkebaek, N.E. Endocrine disruptors and testicular dysgenesis syndrome. *Horm. Res.* **2002**, *57* (2), 43.
15. Melnick, R.; Lucier, G.; Wolfe, M.; Hall, R.; Stancel, G.; Prins, G.; Gallo, M.; Reuhl, H.; Ho, S.-M.; Brown, T.; Moore, J.; Leakey, J.; Haseman, J.; Kohn, M. Summary of the National Toxicology Program's report of the endocrine disruptors low-dose peer review. *Environ. Health Perspect.* **2002**, *110* (4), 427–431.
16. Schmidt, C.W. The lowdown on low-dose endocrine disruptors. *Environ. Health Perspect.* **2001**, *109* (9), 420–421.
17. Yonemoto, J. The effects of dioxin on reproduction and development. *Ind. Health* **2000**, *38* (3), 259–268.
18. Savas, U.; Griffin, K.J.; Johnson, E.F. Molecular mechanisms of cytochrome P-450 induction by xenobiotics: an expanded role for nuclear hormone receptors. *Mol. Pharmacol.* **1999**, *56* (5), 851–857.
19. Waxman, D.J. P-450 gene induction by structurally diverse xenochemicals: central role of nuclear receptors CAR, PXR, and PPAR. *Arch. Biochem. Biophys.* **1999**, *369* (1), 11–23.
20. Straube, E.; Straube, W.; Krüger, E.; Bradatsch, M.; Jacob-Meisel, M.; Rose, H.-J. Disruption of male sex hormones with regard to pesticides: pathophysiological and regulatory aspects. *Toxicol. Lett.* **1999**, *107* (1–3), 225–231.
21. Vinggaard, A.M.; Hnida, C.; Breinholt, V.; Larsen, J.C. Screening of selected pesticides for inhibition of CYP19 aromatase activity in vitro. *Toxicol. In Vitro* **2000**, *14* (3), 227–234.
22. Oliva, A.; Spira, A.; Multigner, L. Contribution of environmental factors to the risk of male infertility. *Hum. Reprod.* (Oxford, England) **2001**, *16* (8), 1768–1776.
23. Herbst, A.L.; Ulfelder, H.; Poskanzer, D.C. Adenocarcinoma of the vagina. Association of maternal stilbestrol therapy with tumor appearance in young women. *N. Engl. J. Med.* **1971**, *284* (15), 878–881.
24. Bibbo, M.; Gill, W.B.; Azizi, F.; Blough, R.; Fang, V.S.; Rosenfield, R.L.; Schuhmacher, G.F.P.; Sleeper, K.; Sonek, M.G.; Wied, F.; Wied, G.L. Follow-up study of male and female offspring of DES-exposed mothers. *Obstet. Gynecol.* **1977**, *49* (1), 1–8.
25. Klip, H.; Verloop, J.; van Gool, J.D.; Koster, M.E.; Burger, C.W.; van Leeuwen, F.E. Hypospadias in sons of women exposed to diethylstilbestrol in utero: a cohort study. *Lancet* **2002**, *359* (9312), 1102–1107.
26. Ehrhardt, A.; Meyer-Bahlburg, H.; Rosen, L.; Feldman, J.; Veridiano, N.; Zimmermann, I.; McEwen, B.S. Sexual orientation after prenatal exposure to exogenous estrogen. *Arch. Sex. Behav.* **1985**, *14* (1), 57–77.
27. Scheirs, J.G.M.; Vingerhoets, A.J.J.M. Handedness and other laterality indices in women prenatally exposed to DES. *J. Clin. Exp. Neuropsychol.* **1995**, *17* (5), 725–730.
28. Garry, V.F.; Tarone, R.E.; Kirsch, I.R.; Abdallah, J.M.; Lombardi, D.P.; Long, L.K.; Burroughs, B.L.; Barr, D.B.; Kesner, J.S. Biomarker correlations of urinary 2,4-D levels in foresters: genomic instability and endocrine disruption. *Environ. Health Perspect.* **2001**, *109* (5), 495–500.
29. Straube, E.; Straube, W.; Krüger, E.; Rose, H.-J. Untersuchungen zur Beeinflussung von Sexualhormonen durch Pestizide bei beruflicher Exposition in der Landwirtschaft und Schädlingsbekämpfung. *Arb. med. Soz. med. Umweltmed.* **2002**, *37* (1), 35–40.
30. <http://www.epa.gov/epahome/research.htm>→EPA_Test_Methods_and_Guidelines→OPPTS_Harmonized_Test_Guidelines→870_Health_Effects_Test_Guidelines (accessed March 2003).

Host-Plant Selection by Insects

Rosemary H. Collier
Stan Finch

Warwick HRI, The University of Warwick, Wellesbourne,
Warwick, U.K.

INTRODUCTION

In natural situations, most plants grow surrounded by a wide range of other highly diverse plant species, and it is in such situations that specialist insects must find their specific host plants.

Many entomologists^[1] have suggested that specialist phytophagous insects find their host plants by orienting to the volatile plant chemicals that are released by, and are characteristic of, specific host plants. While the mechanisms put forward to describe how olfaction regulates host-plant finding seem plausible, Kennedy^[2] indicated in 1977 that most of the mechanisms were based on untested assumptions. The same is still true today. The major problem has been in deciding how to design experiments to show that flying insects can obtain directional cues from plant odors. This has been extremely difficult because of the disruptive air movements around plants, the small amounts of volatile chemicals released, the short distances over which the insect responds, and the closing speed of the insect prior to landing. The current theory, in which the central stage of host-plant finding is based on visual stimuli, helps to explain why experiments to show that olfaction is the crucial component that guides insects to their eventual host plants have so far proved intractable.

NEW THEORY

We developed our theory by simply observing how insects behave. Our theory is based on the facts that phytophagous insects land indiscriminately on green objects, such as the leaves of host plants (appropriate landings) and non-host plants (inappropriate landings), and avoid landing on brown surfaces, such as soil.

In our theory,^[3] we divide host-plant selection into three closely linked stages. In the first stage, the characteristic odors given off by certain plants indicate to dispersing insects that they are flying over suitable host plants. Therefore, the primary effect of plant odors is to stimulate insects to land. Under suitable weather conditions, plant odors may also provide some directional information, but this is of secondary importance. The second important fact contributing to the

new theory is that phytophagous insects rarely land on surfaces that are colored brown and so they avoid landing on soil. As a result, insects that fly over plants growing in bare soil will be stimulated to land on the only green objects available to them—host plants—and so most landings will be “appropriate.” In contrast, insects flying over host plants surrounded by non-host plants will land in proportion to the relative areas occupied by the leaves of the host and the non-host plants, as phytophagous insects do not discriminate between host plants and non-host plants when both are green.^[4] Hence, any landings made on the non-host plant are “inappropriate,” as the plant is not suitable for oviposition. The central stage in host-plant selection, therefore, is based on a combination of “appropriate/inappropriate landings”^[3] and is governed by visual stimuli. The time the insect spends on the leaf of the non-host plant depends upon whether the insect receives acceptable or unacceptable stimuli through its tarsal and gustatory receptors. Once it takes off from a non-host leaf, if the insect is stimulated to land after flying only a relatively short distance, it could land on a host plant. However, the plant on which the insect first lands, even if it is a “host plant,” may not be sufficiently stimulating to arrest the insect and so the whole process will be repeated. If this represented the complete system, it could just be a matter of time before the numbers of eggs laid on host plants growing in diverse backgrounds were similar to those laid on host plants growing in bare soil. However, this does not occur, as there is a second part to the host-plant finding stage.

This second part can be illustrated (Fig. 1) most clearly using data collected on the behavior of the cabbage root fly. Fig. 1 shows that each female cabbage root fly usually makes about four spiral flights. Each time the female lands, it reassesses the suitability of the plant as a site for oviposition. Hence, the female stands a much greater chance of “losing” the host plant in a diverse background as, on average, it repeats the initial appropriate/inappropriate landing procedure a further three times. Observations under laboratory conditions showed that for every 100 female flies that landed on a brassica plant surrounded by bare soil, 36 received sufficient

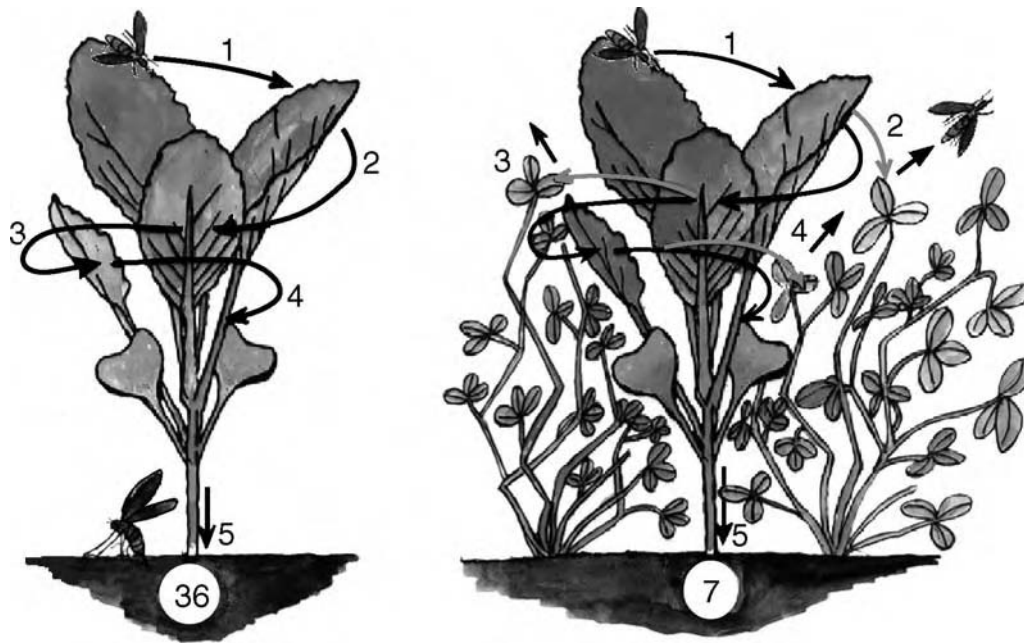


Fig. 1 Schematic diagram to show how diverse backgrounds, here represented by clover (*Trifolium* spp.), influence host-plant acceptance by the cabbage root fly. Numbers represent the four (mean no.) leaf-to-leaf flights made by the fly to ascertain whether the plant is a suitable site for oviposition. (From Ref.^[3])

stimulation to lay eggs. In contrast, only 7 (Fig. 1) out of 100 females that landed on host plants surrounded by non-host plants were stimulated to lay eggs. Fewer flies managed to lay in this situation because, following each spiral flight, a proportion of the flies landed on the surrounding non-host plants. This failure to recontact a host plant after any spiral flight prevented the females from accumulating, at the required rate, sufficient stimulation from the host plant to lay.

HOST-PLANT ACCEPTANCE

Once an insect has landed on a plant, host-plant finding (Stage 2) becomes truly integrated with host-plant acceptance (Stage 3), as the complete system consists of finding and refinding the host plant. The schematic shown in Fig. 2 indicates that, on average, the female cabbage root fly needs to visit four host-plant leaves (2) to accumulate sufficient stimulation to lay. However, a female may only need to visit two leaves of a highly stimulating plant (1) compared to six leaves on a poorly stimulating plant (3). In contrast, other individuals may accumulate sufficient stimuli to keep them searching (4), but not sufficient for oviposition, and so they fly away. A similar outcome is produced when insects visit several leaves (here shown as (3)) but do not accumulate sufficient stimuli in the allotted time to be induced to stay (5). Two other variations

occur when the insects land initially on a stimulating leaf but subsequently land on a non-stimulating leaf. It does not matter whether this leaf is from a host (6) or a non-host plant (7), as anything that interrupts the rate of accumulation of positive stimuli causes the insect to move elsewhere. Finally, the new immigrants may not remain on otherwise-acceptable plants if the plants are already colonized by certain other insect species (see 3).

OTHER CONTRIBUTING FACTORS

The physiological state of the insect, which depends partly on its age and on how long it has been deprived of a suitable oviposition site, has to be superimposed upon this already complex system.^[5] The condition of the plant is also extremely important, as some host-plant species are more highly preferred than others and during periods of rapid growth, many individual plants become highly stimulating to insects.^[6] However, even if the insect and the plant are both in the optimum physiological state, it counts for nothing the moment the insect makes a wrong "choice" and alights on any green object other than a host plant. In practice, the probability of making a wrong choice is reduced considerably when the host plant is highly stimulating, as the insect has to visit fewer leaves. The wide range of other factors involved during host-plant acceptance are reviewed elsewhere.^[7]

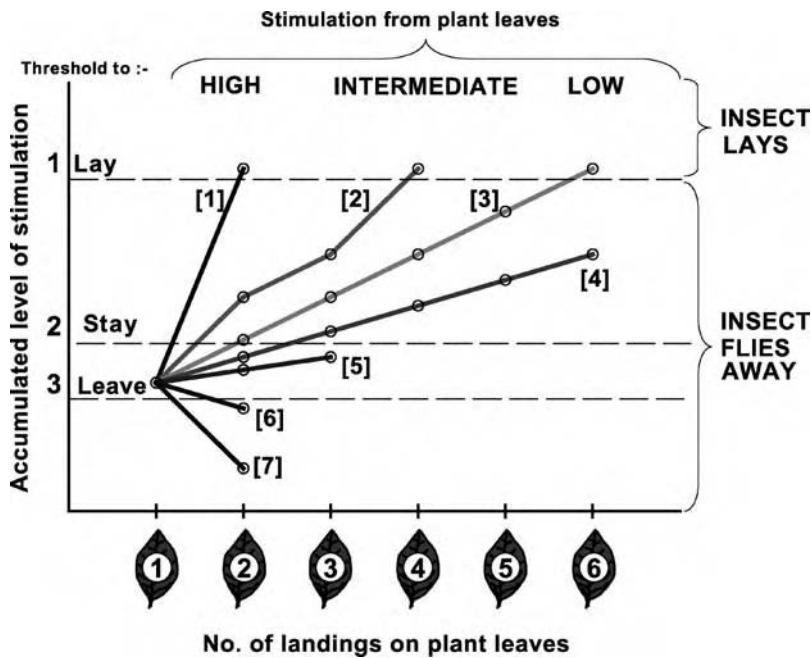


Fig. 2 The number of leaf landings a cabbage root fly may have to make before accepting a plant as a suitable oviposition site or deciding to fly elsewhere. The numbers in brackets represent seven possible variations in the pattern of insect behaviour. (From Ref.^[3])

PRACTICAL CONSIDERATIONS

The effect of diverse backgrounds on host-plant selection by insects simply reflects the numbers of contacts/recontacts the insect has to make to be stimulated to lay. Recent results^[3] showed that the diamond-back moth (*Plutella xylostella*) was the species affected least by diverse backgrounds. This raises the question of whether this moth has become the major world pest of cruciferous crops simply because it has a limited behavioral repertoire and so lays its eggs on more or less the first host-plant leaf it encounters.

From a crop protection point of view, the more non-host plants removed from any crop area, the greater chance a pest insect has of finding a host plant. Hence, our current cultural methods are exacerbating our pest control problems, as “bare-soil” cultivation ensures that crop plants are exposed to the maximum pest insect attack possible in any given locality.

The new theory indicates that it is just the number of green objects surrounding a host plant that reduces colonization by pest insects. Hence, it should not be too difficult to quantify the plant architecture needed in the interrow spaces to reduce pest insect numbers in any given crop.

FUTURE WORK

The theory of “appropriate/inappropriate landings” raises searching questions concerning several aspects of entomological research. These include: 1) whether host-plant volatile chemicals are truly attractants or

simply arrestants for receptive insects;^[8] 2) how the aromatic plants used in “companion planting” produce their effects; and 3) how to select the most suitable non-host plants to grow as intercrops.^[9]

Apart from its impact on pest control situations, the “appropriate/inappropriate landings” theory can also be used to explain why wild host plants growing in natural vegetation are attacked by some individuals of “pest” species but are rarely decimated by them. This raises the question of whether the progeny of the insects that develop on wild host plants remain on them in subsequent generations? If they do, then is this how populations diverge? The answer to this question is of considerable practical importance because, if the insects that colonize wild host plants are in effect separate populations, it should be possible to control certain pest insects by isolating new crops from earlier infestations.

Additional work is required to determine whether the mechanisms used by beneficial insects (predators and parasitoids) to find pest insects are also affected by appropriate/inappropriate landings. If they are, then the suggestions of some researchers that diverse backgrounds have adverse effects on pest insects and no effect on the associated beneficial insects warrant further study.

Irrespective of how this theory is received by researchers studying host-plant attractants, “appropriate/inappropriate landings” does appear to provide a robust description of host-plant selection by insects under a wide range of conditions. While we believe the simplicity of our theory makes it all embracing, only time will tell whether our optimism is justified.

REFERENCES

1. Dethier, V.G. Chemical interactions between plants and insects. In *Chemical Ecology*; Sondheimer, E., Simeone, J.B., Eds.; Academic Press: New York, 1970; 83–102.
2. Kennedy, J.S. Olfactory responses to distant plants and other sources. In *Chemical Control of Insect Behaviour: Theory and Application*; Shorey, H.H., McKelvey, J.J., Jr., Eds.; Wiley: New York, 1977; 67–91.
3. Finch, S.; Collier, R.H. Host-plant selection by insects—a theory based on “appropriate/inappropriate landings” by pest insects of cruciferous plants. *Entomol. Exp. Appl.* **2000**, *96*, 91–102.
4. Prokopy, R.J.; Owens, R.D. Visual detection of plants by herbivorous insects. *Annu. Rev. Entomol.* **1983**, *28*, 337–364.
5. Barton Browne, L. Physiologically induced changes in resource oriented behavior. *Annu. Rev. Entomol.* **1993**, *38*, 1–25.
6. Finch, S. Chemical attraction of plant-feeding insects to plants. In *Applied Biology V*; Coaker, T.H., Ed.; Academic Press: London, 1980; 67–143.
7. Simpson, S., Mordue (Luntz), A.J., Hardie J., Eds.; *Proceedings of the Tenth International Symposium on Insect-Plant Relationships*; *Entomol. Exp. Appl.*; 1999; Vol. 91, 1–265.
8. Kennedy, J.S. The concepts of olfactory “arrestment” and “attraction”. *Physiol. Entomol.* **1978**, *3*, 77–89.
9. Cromartie, W.J., Jr. The environmental control of insects using crop diversity. In *CRC Handbook of Pest Management in Agriculture*; Pimentel, D., Ed.; CRC Press: Boca Raton, Florida, 1981; 1, 223–251.

Household Pest Management: Insects and Mites

William H. Robinson

Urban Pest Control Research, Christiansburg, Virginia, U.S.A.

INTRODUCTION

A large number of insects and mites occur in the human environment. Many of these are considered pests because of their economic, medical, or aesthetic influence on the quality of life. This group of arthropods adapted to the habitats and conditions created when natural environments were altered or agricultural environments further developed to provide living and recreation space for people and pets. The human environment includes metropolitan urban and suburban areas. This environment can be divided into two groups of distinct habitats: domestic and peridomestic. Domestic habitats include the variety of plants and animals and stored food and fabric materials found indoors. Peridomestic habitats are the soil, ornamental trees, and shrubs around the outside of structures, and the recreation or green zones in urban and suburban areas. In some locations, human structures and activity may interface with the natural environment, and in other locations activity may interface with the agricultural environment. In all these locations, there are arthropods that interact with people and pets.

Some of the arthropods that have adapted to domestic habitats may no longer exist in the natural populations outside the house. Examples of these include beetles and moths that infest stored food and fabric, some cockroaches that infest kitchens and bathrooms, and species of household mites. These arthropods have been associated with humans or the household environment for a long time, and the populations that form natural reservoirs are rare or have disappeared. A large number of arthropods have adapted to living in peridomestic habitats. They find food and harborage in habitats in the soil and ornamental trees and shrubs that have been planted outside structures. Examples of peridomestic pests include ants that nest in the soil, butterflies, moths, and various sap-sucking insects associated with plants; the bees and wasps that nest below or above ground; and flies and beetles that feed on decaying organic matter. Natural populations of many peridomestic species also occur in undisturbed areas or natural areas, or in agricultural areas that interface with the human environment.

Pest status for insects and mites in and around the household environment may have an aesthetic, medical, or economic basis. Aesthetics is an important basis

for controlling pests in the human environment, and for many arthropods it is the primary reason. For example, the mere presence of house flies, silverfish, or house centipedes indoors can be unacceptable to some people. The presence of cockroaches in kitchens and bathrooms may be socially unacceptable. Spiders, bees, wasps, ants, and fleas can inflict a painful bite or sting. Some individuals are hypersensitive to these insects or to the allergens in the feces or body fragments. From an economic standpoint, several species of beetles and moths can taint food or damage clothing. Regardless of the medical or economic influence, the pest status for most domestic and peridomestic arthropods is based on their presence, individually or in large numbers, around people or pets.

The strategies and goals typically used for pests in the agricultural environment may not be applicable in the household environment. In the human living space, pest control objectives include managing pest infestations when this is feasible and acceptable, but the elimination of infestations when the pest presents an unacceptable risk. For the majority of peridomestic pest species, reducing or managing the occurrence of populations may be sufficient. This may be accomplished by altering habitats, removing critical resources, like food, or direct chemical control or trapping tactics. For domestic species, whether they are represented by actual infestations or only seasonal invaders, elimination is usually the goal. This may be accomplished by preventing access to the structure or direct chemical control tactics.

MANAGEMENT STRATEGIES FOR PERIDOMESTIC PESTS

The habitats utilized by peridomestic pests include the various types of soil and vegetation surrounding houses and other structures. Here, the variety of organic and inorganic substrates is matched by an equal variety of insects and mites that utilize them. The categories of insects and mites associated with the peridomestic environment include several species of social insects, nuisance and solitary species, and species that overwinter in or around structures.

The presence of pest reservoirs in the human environment or in natural or agricultural environments

place limitations on control of some peridomestic pests. These insects are important to the ecological dynamics of soil habitats and to the arthropod fauna of ornamental plants. Their control or elimination may be detrimental to the environment. The objectives of management strategies for the majority of these insects are to limit pest populations or prevent individuals from foraging near people or occurring indoors.

Solitary Insects

There are several species of solitary bees (*Dialictus* spp., *Lasioglossum* spp.) that are a nuisance by their nesting in bare soil around houses and in recreational areas. There are species of flies, including the house fly (*Musca domestica*) and blow flies (*Phaenicia sericata*, *Phormia regina*) that can be annoying by their presence indoors. Non-chemical strategies for reducing these insects include removing the sources of attraction and breeding. Covering garbage can reduce the presence of house and blow flies, and planting grass to cover bare soil can reduce the presence of solitary bees. Chemical methods of reducing the presence of these insects include the use of traps and baits for flies, and applying insecticides to nesting sites of solitary bees.

Social Insects

Yellowjackets (*Paravespula* spp.), umbrella wasps (*Polistes* spp.), honey bees, several species of ground-nesting ants (*Acanthomyops* spp., *Formica* spp., *Solenopsis* spp.) occur around or in buildings. The medical importance of wasps dictates that colonies be eliminated when they pose an immediate threat to people. Modern control methods include the use of liquid and aerosol insecticides and toxic baits that target colonies located close to dwellings. There are traps that can effectively reduce the number of yellowjackets foraging close to dwellings without eliminating entire colonies. Colonies of ant species that nest outdoors but often forage indoors can be reduced with the use of baits placed around the perimeter of structures.

Overwintering Insects

Several insects overwinter around the outside and inside of structures in the urban environment. The most common of these are the boxelder bug (*Boisea trivittatus*), cluster fly (*Pollenia rudis*), and the Asian ladybird beetle (*Harmonia axyridis*). The pest status of these and insects with similar habits is based primarily on their presence in large numbers, and to some extent on their activity indoors during the winter months as they hibernate. Control strategies are based

on attempts at exclusion, to a limited extent on sticky traps, and on spraying the outside of houses with insecticides. When boxelder bugs are persistent, removing the female trees is often considered. This is not recommended and is rarely successful, because these insects can feed on other maple trees.

PREVENTION AND ELIMINATION STRATEGIES FOR DOMESTIC PESTS

The habitats utilized by the infestation of domestic pests include the materials, stored food, and fabric that have characterized the human living space for thousands of years. These arthropods adapted to substrates and habitats unique to household environments, and natural reservoirs of these species are not known. Re-infestation often occurs from other domestic habitats. The major categories of insects and mites associated with the domestic environment are infesting insects and invading insects. Infesting pests include species that utilize household materials and substrates, and that reproduce and have multiple generations indoors. Invading species do not reproduce indoors, but are there occasionally during the year.

Infesting Insects

Control strategies for infesting insects begin with changes in the environment resources that provide for the long-term survival of pest populations. For cockroaches these actions include reducing the amount of food, water, and harborage available. Infestations of stored food pests such as psocids, flour beetles, and moths may be eliminated or controlled by storing bulk materials in sealed containers, and vacuuming scattered flour, meal, and other flour-based foods from cabinets. For clothes moths and carpet beetle infestations, the strategies include cleaning to remove existing larval stages, then storing clothing in sealed containers, and cleaning the immediate habitat. Pheromone-based sticky traps for Indian meal moth (*Plodia interpunctella*), clothes moth (*Tinea* sp., *Tineola* sp.) and for many stored-food beetle and moth pests are an effective monitoring and control strategy for small household infestations.

Removal of limiting necessary resources may be difficult or impractical for pests such as silverfish and house centipedes. These insects move around the household and may be able to find food and harborage in a number of sites. Sticky traps placed in sites they frequent may provide limited control. For seasonal and short-term infesting pests, such as fruit flies and fungus gnats, source elimination is the most effective control strategy.

Invading Insects

Strategies for insects that invade structures from the outside include physically blocking their access points, and changing the environmental conditions favoring their presence. These insects, mites, and spiders may be linked to an abundance of harborage or food around the immediate perimeter of the structure or the surrounding grounds. Millipedes and clover mites may utilize the turfgrass, and reducing the thatch layer or planting different varieties may help reduce their numbers. Field crickets, earwigs, sowbugs, and centipedes find favorable harborage and food in organic mulch surrounding buildings or covering ornamental plant beds. Mulch kept as dry as possible or limited to well-drained sites will reduce its attractiveness to these animals.

Limiting the use or wattage of outdoor lights can reduce the insects coming to the house perimeter at night, and in turn reduce the presence of spiders, scorpions, and other predators. Managing the use of outdoor lights may also reduce the incidence of sod webworm adults, carabid beetles, and some other nocturnal insects that often enter houses after first collecting on door and window screens. Many of these arthropods enter houses around widows and door thresholds. Reducing the gaps around ground-level doors, and windows can help prevent the entry of many crawling and flying insects. Garbage and trash cans kept close to doors and windows may contribute to the house flies and fruit flies that are seasonal pests indoors. Cleaning these containers regularly and maintaining tight-fitting lids can reduce their attractiveness to pests such as flies and yellowjackets.

FUTURE PEST MANAGEMENT STRATEGIES

Insects and mites in the household continue to be an important aspect of the quality of life. Concern for exposure to pesticides and the pest status of arthropods results in the use of combinations of chemical and non-chemical methods for peridomestic and domestic pests, and effective pest management and prevention methods for most peridomestic pests. Use

of predators and parasites for managing household insect and mite pests is generally not practical or effective for these pests.

Peridomestic Habitats

Pest reservoirs in urban green spaces and scattered pest populations in the soil and ornamental plants and trees in suburban areas will continue to provide individuals or colonies of arthropods that enter houses or other structures. Efforts to manage stinging insects and seasonal pests such as earwigs, boxelder bugs, Asian ladybird beetles, and cluster flies will depend on the use of baited traps and selected chemical applications. Preventing insects from entering structures or limiting colonies of bees and wasps will be the most effective strategies. Management strategies will be the most effective, because eliminating many of these pests may not be possible due to the pest populations in adjacent natural areas.

Domestic Habitats

Decreased use of liquid and dust insecticides in favor of baits and on-animal applications to control cockroaches and fleas may result in increased infestations of ants, silverfish, and stored-food insects. Continued development of pheromone-based traps, species-specific baits, or baited sticky traps will provide for pest management programs that emphasize monitoring and eliminating pest populations. In spite of concern for pesticide exposure and residues indoors, pest elimination, and not management is likely to remain the primary objective for indoor pests. Low-concentration insecticides applied as aerosols, liquid sprays, dusts, and species-specific baits will provide limited exposure and desired control.

BIBLIOGRAPHY

Robinson, W.H. *Urban Entomology*; Chapman & Hall: London, 1996.

Hoverflies: Indicators of Sustainable Farming and Potential Control of Aphids

Daniele Sommaggio

Biostudio, Velo d'Astico (VI), Italy

Giovanni Burgio

*Dipartimento di Scienze e Tecnologie Agroambientali-Entomologia, Alma Mater Studiorum
Università di Bologna, Bologna, Italy*

INTRODUCTION

Usually called hoverflies by Europeans and flower flies by Americans, Syrphidae is a Diptera family. To date, more than 6000 species have been described but more than 14,000 may exist. The beauty of several species has stimulated many authors to collect and study them, but interest in Syrphidae is also extended to their importance in agriculture and, more recently, in biodiversity conservation.

More than one-third of Syrphidae have predatory larvae, mainly aphidophagous ones, and are important in controlling pest population. In addition, larvae with similar trophic habitus (e.g., phytophagous, predatory, saprophagous) show very different environmental requirements; for this reason, hoverflies have been suggested as effective bioindicators to evaluate nature conservation.

In the present paper, the authors will review the importance of Syrphidae in agroecosystems. The attention will be focused mainly on three topics: the role of Syrphidae in aphid control; agroecosystem management in enhancing hoverfly population, and, finally, their use as bioindicators, with special reference to agroecosystems. In the first part, a short survey about techniques to collect and study hoverflies will be provided to give some practical information to develop a monitoring plan.

COLLECTING AND STUDYING HOVERFLIES

Investigations of larval biology provide essential data for managing the many common species that are economically useful and the few that are pests. Practical information about collecting and preserving morphology and identification has been recently published in practical manuals. Larvae of predatory species can be manually collected on vegetation, for example, by hand-searching of aphid colonies. Larvae can also be collected by beating of trees and shrubs with beating

tray or by removing organs of plants and placing in clear plastic bags.

Many sampling methods are available to collect and monitor adults as follows:

- **Malaise traps:** The use of Malaise trap has been suggested as standard method to collect hoverflies. This method is a standard system to collect adults, to study fenology, and to compile list of species; on the other hand, the method is time-consuming, especially when using replicated traps. Malaise traps, if well managed, can provide, in some cases, data for quantitative analysis.
- **Hand-net:** This method is affected by the ability of collector and environmental conditions. It is considered a subjective method of sampling; hand-net can supply detailed faunistic lists and provide complementary data to Malaise traps. By using both Malaise traps and hand-net, it is possible to obtain a wider spectrum of the Syrphidae fauna in a site.^[1]
- **Chromotropic traps:** Syrphidae adults are attracted by yellow and white and the cromotropism of adults can be exploited to collect and monitor specimens. In order to catch adults, water or glue can be used. This is a practical and low-cost method to monitor Syrphidae population and to provide faunistic lists, but more frequent visits are needed, especially if water is used.

SYRPHIDAE AS POTENTIAL CONTROL OF APHIDS

All species in the Syrphinae subfamily and in the Pipizini tribe are predators on soft body insects, such as aphids, coccids, and psyllids, although a few specialize on other types of prey such as microlepidopteran caterpillars, noctuid larvae, tenthredinid larvae, ant broods, chrysomelid beetle larvae, flies, and mites. Recently, the range of prey has been revised,^[2] providing a worldwide data bank.

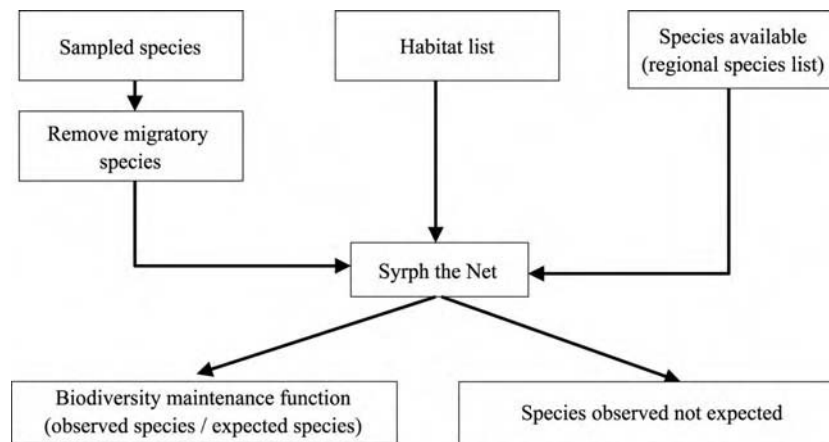


Fig. 1 Syrph the Net procedure for environmental analysis. “Sampled species” can be obtained by sampling; “habitat list” is codified in accordance with CORINE system and must be recorded during sampling visit. A “regional species” list must be available to use Syrph the Net. (From Ref.^[9].)

Syrphids can be effective in conservation biological control, and their role in limiting aphid population has been quantified both in laboratory^[3] and field tests.^[4] Several factors can strongly affect hoverfly population and, consequently, the control on aphid. Faunistic studies in Northern Italy demonstrated that percentage of parasitization of Syrphid larvae by Hymenoptera parasitoids can be very high on crops like alfalfa (82%) and wheat (49%);^[5] this factor could be responsible for their population density variability. On the other hand, the interactions plant–herbivore–predator are very complex; further studies are needed to clarify many aspects of these tritrophic interactions.^[6]

Recently, hoverflies have been tested also to increase biological control after rearing and releasing techniques. This method has been proven to be effective in limiting, for example, *Aphis gossypii* populations in greenhouse.^[3] Inundative release of eggs or larvae would be very time-consuming for the grower, but several releases of gravid females would be a quick and simple task. Prereproductive females are not a suitable stage to release because they are inclined to disperse and, in sunny weather, they would leave the glasshouse through the vents. The aphidophagous *Episyrphus balteatus* is now being mass-reared by a biofactory and it is commercialized at pupal stage for releases on vegetables in greenhouse.

FIELD STRATEGIES TO ENHANCE HOVERFLY POPULATIONS IN AGROECOSYSTEMS

The pest agent control are hoverfly larvae, while adults are all pollinivore. Introducing flower strips can easily increase adult population as recently showed.^[7] The question is how much these adults can colonize the adjacent field, increasing predatory pressure on crop

pests. Despite their good ability in flying, e.g., some species regularly migrate in Europe, adult mobility seems to be limited to adult feeding and oviposition sites. In agroecosystems, for example, introducing *Phacelia* strips on margin field can increase *Melanostoma* population only at a distance shorter than 50 m; females with *Phacelia* pollen in the gut are usually found within 25–30 m from flower strips.^[8] Increasing adult population on field margin does not necessarily increase aphidophagous larvae on the crops. It is necessary to clarify the effect of margin vegetation focusing not only on adults but mainly on larvae, the real pest agent control. Few researchers have studied the influence of margin vegetation on the density and spatial aggregation of larvae. In some cases, crops with more diverse field margins have been observed to support higher level of larvae, but it is important to consider all parameters such as the number of eggs and larvae/shoot or the phenology of larvae distribution. In fact, larvae can be more abundant near hedgerows at the beginning of the year, when control effect can be stronger.^[8]

HOVERFLIES AS INDICATORS OF BIODIVERSITY

Conservation of biodiversity has become a primary goal in any environmental planning and management. In natural ecosystems, research is focused on hoverfly biodiversity; however, in agroecosystems, attention is usually drawn to aphidophagous species and their density to increase pest control, but little attention is paid to a general conservation approach.

Hoverfly family has been suggested as a good indicator of biodiversity by many authors,^[8,9] and several practical cases clearly show the utility of this family

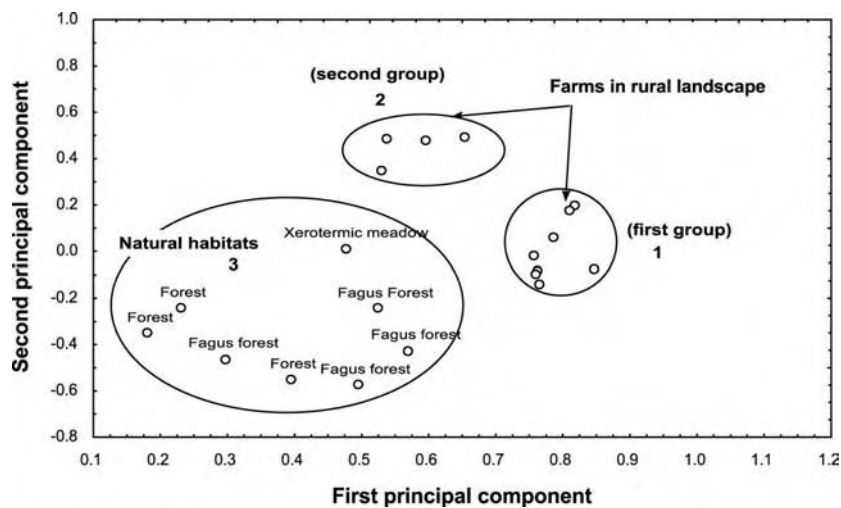


Fig. 2 Ordination of different sites by principal component analysis (PCA) performed on faunistic lists of Syrphidae collected by hand-net (data analyzed by a presence/absence matrix). PCA forms three main groups: 1) farms in rural landscape characterized by high plant diversity and connected ecological corridors (first group); 2) farms in rural landscape with low plant diversity and high anthropic impact (second group); and 3) natural habitats including forests. All the sites are ordered according to the anthropic impact and landscape management.

in environmental analysis.^[9,10] Recently, for Atlantic Europe, a technique called Syrph the Net has been developed, which can be used as a tool in measuring biodiversity.^[9] Fig. 1 simplifies the use of Syrph the

Net. This technique can be extended to other regions.^[11] Syrph the Net can be used not only to evaluate specific habitat but also to simulate human pressure in environments. Recently, Syrph the Net has been used as

Table 1 Syrphidae species number in different areas in Northern Italy

| Regional area | Locality | Ecosystem | N. spp. | Method |
|---------------|-----------------------------|-------------------------------------|---------|--------|
| Alps | Summano mountain | Fagus wood | 87 | M, E |
| Alps | Pastello mountain | Young Fagus Wood | 54 | M, E |
| | Pastello mountain | Xerothermic meadows | 36 | M, E |
| Alps | Lower part of Adige Valley | Fraxinus ornus wood | 64 | M, E |
| Alps | Pasubio mountain | Mixed forest | 87 | M, E |
| Appennines | Campigna | Mixed forest | 92 | E |
| Appennines | Castiglione dei Pepoli | Abies wood | 73 | E |
| Po Plain | Ferrara town | Suburban park | 43 | M, E |
| Po Plain | Campotto (Ferrara province) | Alluvional wood | 48 | M, E |
| | Campotto (Ferrara province) | Wetland | 24 | M, E |
| Po Plain | Mesola (Ferrara province) | Deciduous wood | 39 | M, E |
| Po Plain | Verona province | Ochards | 33 | C, E |
| Po Plain | Bologna province | Orchard and vegetables ^a | 31 | M, E |
| Po Plain | Bologna province | Orchard and vegetables | 27 | E |
| Po Plain | Bologna province | Orchard | 16 | E |
| Po Plain | Bologna province | Arable crops | 27 | E |
| Po Plain | Bologna province | Arable crops | 24 | E |
| Po Plain | Bologna province | Arable crops | 19 | E |
| Po Plain | Ferrara province | Arable crops | 15 | E |
| Po Plain | Ferrara province | Arable crops | 10 | E |

M = Malaise traps; E = Hand-net; C = Chromotropic traps.

^aThe farm is neighboring to a wood inside a protected area.

predictive method in planning agroecosystem management with the goal to increase biodiversity.^[10]

The effect of different farming systems on hoverfly fauna has been revised.^[9] Due to Syrphidae mobility, landscape texture can strongly affect hoverfly population in agroecosystems;^[8] more research is needed to better clarify the effect of landscape on hoverfly fauna in agroecosystem. Fig. 2 shows that habitats, including natural and rural sites, can be ordered according to the anthropic impact and landscape management using faunistic lists of Syrphidae, confirming the efficiency of these insects as bioindicators.

Despite the lower landscape biodiversity, Syrphidae can show a good species range also in agroecosystems. In Table 1, different areas in Northern Italy have been compared regarding the number of Syrphidae species. Agroecosystems with different management can support a wide range of species; in some cases, a number of species similar to more natural ecosystems have been noticed. In addition, also rare species can be found in well-managed agroecosystems; for example, in farms characterized by hedgerows and ecological corridors in Po Plain, Italy, rare species, such as *Milesia crabroniformis*, can be collected. A rational agroecosystem management can allow the development of a richer fauna; attention should be paid not only to useful insects in pest control but also to biodiversity as a whole. As a large part of terrestrial ecosystems is cultivated, efforts should be made to preserve not only natural areas but also biodiversity in agroecosystems.

CONCLUSION

An increased interest in Syrphidae has been recently noticed, as suggested by the regular publication of a journal (*Volucella*) in 1995 and international meetings since 2001. An increase in research will surely improve our knowledge of these insects and their biology.

Agroecologists should regard this growing interest as a propitious event because a sustainable farming system cannot disregard hoverfly, either as pest agent control or for biodiversity conservation.

REFERENCES

1. Sommaggio, D.; Burgio, G. Role of Diptera Syrphidae as landscape indicators: analysis of some case studies in Northern Italy. Landscape management for functional biodiversity. IOBC WPRS Bull. **2003**, *26*, 145–150.
2. Rojo, S.; Gilbert, F.; Marcos-Garcia, M.A.; Nieto, J.M.; Mier, M.P. *A World Review of Predatory Hoverflies (Diptera, Syrphidae: Syrphinae)*; CIBIO Ediciones: Alicante, Spain, 2003.
3. Chambers, R.J.; Adams, T.H.L. Quantification of the impact of hoverflies (Diptera: Syrphidae) on cereal aphids in winter wheat: an analysis of field population. J. Appl. Ecol. **1986**, *23*, 895–904.
4. Tenhumberg, B. Estimating predatory efficiency of *Episyrphus balteatus* (Diptera: Syrphidae) in cereal fields. Environ. Entomol. **1995**, *24*, 677–691.
5. Ferrari, R.; Burgio, G.; Pozzati, M.; Zwakhals, K. Segnalazioni di parassitoidi di Ditteri Sirfidi in Emilia-Romagna. Inform. Fitopatol. **1998**, *4*, 76–80.
6. Sadeghi, H.; Gilbert, F. Aphid suitability and its relationship to oviposition preference in predatory hoverflies. J. Anim. Ecol. **2000**, *69*, 771–784.
7. Landis, D.A.; Wratten, S.D.; Gurr, G.M. Habitat management to conserve natural enemies of arthropod pests in agriculture. Annu. Rev. Entomol. **2000**, *45*, 175–201.
8. Sommaggio, D. Syrphidae, can they be used as environmental bioindicators? Agric. Ecosyst. Environ. **1999**, *74*, 343–356.
9. Speight, M.C.D., Castella, E., Obrdlik, P., Ball, S., Eds.; *Syrph the Net, the Database of European Syrphidae*; Syrph the Net Publications: Dublin, 1997–2003; 1–38.
10. Speight, M.C.D.; Good, J.A.; Castella, E. Predicting the changes in farm Syrphid faunas that could be caused by changes in farm management regimes (Diptera, Syrphidae). Volucella **2002**, *6*, 125–137.

Immune Deficiency Effects

Claudio Colosio

International Centre for Pesticide Safety, Busto Garolfo, Italy

INTRODUCTION

The use of pesticides is increasing worldwide, and concern for possible health effects arising from prolonged, low-dose exposure is growing. Immunotoxicity is also included among the possible effects of pesticides. In spite of experimental evidence showing, in some cases, immunosuppression, available data never show a clear immunosuppressive effect in man under low-dose exposure conditions except for slight laboratory changes that need to be further investigated.

IMMUNE DEFICIENCY EFFECTS IN LABORATORY ANIMALS

The immune system is able to recognize and neutralize potentially harmful agents, conferring to the organism resistance to infectious and malignant diseases. The immune function is characterized by the interaction between complex sets of cellular and chemical components, and its action is based on the capacity of recognizing the “self” and the “non-self” in the organism. An alteration of the normal immune function may have two types of consequence: The first is a reduction in the immune activity, which can evolve into immune deficit and increased susceptibility to infectious diseases and neoplasms. The second is an enhancement of the normal immune response, which can evolve into allergy and autoimmunity.^[1]

In some cases, chemical substances may cause alterations in the normal immune function. This kind of activity is defined as “immunotoxicity.” Immunotoxicity data on chemicals can be obtained either from experimental studies, carried out on laboratory animals or in vitro cultures, or from field studies carried out on exposed subjects. As for pesticides, some laboratory data showing immunotoxic effects are available. Immunosuppressive effects have been observed in the laboratory studies of some organophosphorous (OP) compounds (parathion, methylparathion, malathion, and *o,o,s*-trimethylphosphorothioate), organochlorines (OC) (DDT, mirex, hexachlorobenzene, dieldrin, chlordane, and pentachlorophenol (PCP)), carbamates (carbofuran and aldrin, the latter with no univocal data), organotin compounds (triphenyltin hydroxide, and tributyltin oxide), and pyrethroids (deltamethrin

and α -cypermethrin). The evidence of an immunosuppressive effect exerted by *o,o,s*-trimethylphosphorothioate, a production contaminant of some OP formulations namely malathion,^[2,3] suggests that, when pesticides’ immunotoxicity is being evaluated, inert ingredients, as well as possible impurities, should also be taken into account.

IMMUNOTOXIC EFFECTS IN MAN

Despite the results of experimental studies showing the effects caused by pesticides to the immune system, only a few studies have been carried out on humans, and suggested immune deficiency effects. Available data are depicted in Table 1.

OP Compounds

Impairment of neutrophil chemotaxis and adhesion has been observed among workers involved in OP compounds production (chlorfenvinphos, trichlorfon, malathion, dichlorvos, fenitrothion, and phormothion). Upper respiratory infections are more frequent in these workers compared with the control group, and the rate of recurrence proved to be dependent on the duration of exposure.^[4] A decreased percentage of lymphocytes T-helper and of the activation marker CD5, probably resulting from an increase in CD26, were observed in a group of subjects exposed to chlorpyrifos. Some of these workers expressed multiple-organ symptoms (flu-like illness, upper and lower respiratory symptoms), and also atopy and antibiotic sensitivity were increased, but none of the subjects suffered from a major health impairment.^[5] These data suggest a slight immunotoxic effect in man. The possibility that such an effect may have been caused by the immunotoxic contaminant *o,o,s*-trimethylphosphorothioate must be taken into account at least for malathion and fenitrothion.

OC Insecticides

An impairment of neutrophil function has been observed in workers involved in the manufacture of DDT and

Table 1 Pesticides possibly able to cause immune deficiency changes in man under occupational and/or environmental exposure

| Active ingredient | Observed change | References |
|---|--|------------|
| OP compounds chlorfenvinphos, trichlorfon, malathion, dichlorvos, fenitrothion, phormothion | Impairment of neutrophil function Increased susceptibility to infections | [4] |
| Chlorpyrifos | Decrease of T-helper and CD5 Increase of CD26 | [5] |
| DDT and hexachlorocyclohexane | Impairment of neutrophil function Increased susceptibility to infections | [6] |
| Chlordane | Increase of cortical thymocytes Decrease of: T-helper lymphocytes, CD45RA/T4, proliferative response to mitogens, antibody dependent cell-mediated cytotoxicity Increased susceptibility to sinusitis and bronchitis | [9] |
| Aldicarb | Decrease of helper/suppressor ratio | [8] |
| Phenoxy herbicides (2,4-D and MCPA) | Decrease of T-helper, T-suppressor cytotoxic, natural killer, and lymphocyte proliferative response to mitogens | [10,11] |
| Pentachlorophenol | Decrease of lymphocyte proliferative response to mitogens | [12] |
| | Increase of blood monocytes, IL8 serum levels | [14] |
| | Decrease of lymphocyte proliferative response to mitogens, helper/suppressor ratio, T-helper lymphocytes | [15] |
| | Increase of T-cells activation markers and NK activity (only in females), non-organ-specific autoantibodies | [13] |
| | Decrease of T-helper-inducers, lymphocyte proliferative response to mitogens, serum immunoglobulins | |
| | Presence of immature lymphocyte in blood Increase in low grade infections in exposed subjects | [16] |
| Organotin compounds (acute poisoning case) | Impairment of neutrophil function | [17] |

hexachlorocyclohexane (HCH). Concomitantly, these workers suffered from an increased susceptibility to infections, possibly related to immune deficit.^[6] However, when occupational exposure to HCH alone was taken into account, a statistically significant increase in M-immunoglobulins serum concentration was observed,^[7] suggesting, contrary to the previously described study, an enhancement of the immune response.

In a study on a group of subjects living in houses previously treated with chlordane for termite control, only the incidence of respiratory infectious diseases (sinusitis and bronchitis) was measured, but not on individual immune parameters. The study showed a dose-response relationship between chlordane concentrations in the ambient air and frequency of bronchitis and sinusitis in the exposed subjects.^[8] A second study, carried out on subjects occupationally and environmentally exposed to chlordane, showed a significant increase in cortical thymocytes, decreased frequency

of the suppressor-inducer phenotype CD45RA and T-helper lymphocytes, elevated light chain frequencies on B-cells, decreased proliferative response to mitogens, and depressed antibody-dependent cell-mediated cytotoxicity—thus suggesting a diminishing quality in the immune response.^[9]

Carbamates

Altered numbers of CD8-cells and decreased helper/suppressor ratio were observed in a group of women chronically exposed to aldicarb through ingestion of aldicarb-contaminated drinking groundwater. Helper/suppressor rate was negatively correlated with the average daily aldicarb ingestion.^[10] These findings were confirmed by a follow-up study.^[11] These immune changes, which suggest a slight immunosuppression, were the only alterations observed in these subjects.

Table 2 Example of a tier approach for testing immune system in humans

| Tier | Recommended tests |
|------|---|
| 1 | Serum and cellular parameters Complete and differential blood cells count, Immunoglobulins (IgG, IgA, IgE, IgM), complement factors, rheumatoid factor, non-organ-specific antibodies: ANA, SMA & AMA Lymphocyte subpopulations: CD3: T-cells, CD4: T-helper, CD 8: T-suppressor/cytotoxic, CD20: B-cells, CD25-DR: T-cell activated, CD16, CD57: natural killer, CD3/HLA-DR: T-cell activated, CD4/CD8 ratio Functional parameters: Skin prick test, or antigen specific IgE ELISA or RAST tests Immunisation after vaccination with an antigen to which no prior exposure has occurred |
| 2 | Lymphocyte proliferative response to mytogens (phytohaemoagglutinin, Anti-CD3 monoclonal antibody, ionomycin); polyclonal immunoglobulin production in vitro: basal level and after pokeweed stimulation |
| 3 | In vitro cytokine production, basal and after stimulation: TNF, IL1, IL2, IL6, IL4, IL5, IL6, IL10, IL12, IL13, IFN- γ , TGF-B Soluble receptors of cytokines in blood Soluble membrane markers in blood/plasma m-RNA for specific cytokines |

Note: The testing strategy implies that the tests to be performed must be selected on the basis of the available immunotoxicity data and the investigated effect(s).

Phenoxy Herbicides

A group of 10 farmers involved in the application of a commercial formulation containing 2,4-dichlorophenoxyacetic acid (2,4-D) and 4-chloro-2-methylphenoxyacetic acid (MCPA) was studied through the comparison between selected immune parameters measured before and after a 12-day period of exposure. The study showed a significant reduction in T-helper, T-suppressor cytotoxic, and natural killer cells, together with a statistically significant reduction in the lymphoproliferative response to mitogen stimulation.^[12] Furthermore, the described immune alterations were the only abnormal findings observed in these subjects.

PCP

A study carried out on subjects living in PCP-treated log houses, showed elevated frequencies of activated T-cells and T-cell activation markers CD26, with a reduction in the proliferative response of blood lymphocytes to mitogens.^[13] In a study carried out by Colosio and coworkers, which showed mean PCP plasma concentration of 200 $\mu\text{g/l}$, on a group of 32 subjects involved in wood treatment with a PCP containing solution, a moderate but significant decrease in the blastogenetic response to 5% phytohemagglutinin (PHA) was observed in the group with a prolonged exposure to the active ingredient.^[14] According to Colosio, Daniel showed in 1995^[15] an impairment of lymphocyte stimulation responses in a group of subjects exposed to PCP for more than 6 months. The likelihood of impairment occurrence was higher in those subjects showing mean blood PCP levels of 10 $\mu\text{g/l}$ or higher.^[15] Immunotoxicity of PCP has

been observed since 1980, when Klemmer and coworkers showed an increased occurrence of immature lymphocytes in the peripheral blood of subjects involved in wood treatment with a PCP-containing formulation.^[16] Altogether, these findings confirm that PCP is able to cause slight immune deficiency effects.

Organotin Compounds

The following is an account of a reported acute occupational triphenyltin acetate poisoning case: A poisoned subject showed a strong impairment of neutrophil function (reduction of the normal increase of actin polymerization after stimulation with a chemotactic peptide), and the recovery was correlated with the progressive reduction of tin concentrations in body fluids.^[17]

CONCLUSIONS

Few immunotoxicity studies have been addressed to humans, and few of them allow the collection of data should help to define dose-effect and dose-response relationships. None of the studies at present shows clear immunotoxicity due occupational or environmental exposure. These data confirm that evidence of immune suppression by chemicals in humans is considerably less well established than evidence of allergy, although there is a public perception that chemicals generally cause immunosuppression.^[18]

However, the mild changes observed in some studies need to be further investigated, in order to define their prognostic significance in prolonged, low-dose exposure.

Prevention of immune effects should be carried out in the pre-marketing phase (before a pesticide is placed into the market), through the toxicological evaluation of both active ingredients and commercial formulations. The immunotoxicity screening should be carried out based on a tier approach: in case of evidence (or even suspect) of immunotoxicity at the first-level evaluation, adequate further investigation should be carried out. The evidence of clear immunotoxicity in laboratory studies should be carefully evaluated in the decision process of authorisation for use. As for the substances already in use, in case of suspected immunotoxicity additional laboratory studies are recommended, as well as field studies on exposed subjects. Also field studies should be based on a tier approach, according to the testing strategies suggested by reference organizations.^[1] An example of tiered testing strategy is provided in Table 2. The evidence of immunotoxicity in man should bring about the decision of restricting the use, or even banning the use of immunotoxic compounds. Industry should help these preventive activities, refrain from asking the authorization for the use immunotoxic compounds, or voluntary withdraw from the market compounds that are able to cause immune deficiency effects.

REFERENCES

1. IPCS (International Programme on Chemical Safety). *Principles and Methods for Assessing Direct Immunotoxicity Associated with Exposure to Chemicals*. Environmental Health Criteria, World Health Organisation, International Programme on Chemical Safety: Geneva, 1996; Vol. 180.
2. Devens, B.H.; Grayson, M.H.; Imamura, I.K.; Rodgers, K.E. *O,O,S*-trimethylphosphorothioate effect on immunocompetence. *Pestic. Biochem. Physiol.* **1985**, *24*, 251–259.
3. Descotes, J. Immunotoxicity of Pesticides. In *Immunotoxicology of Drug and Chemicals*; Elsevier: Amsterdam, 1988; 337–363.
4. Hermanowicz, A.; Kossman, S. Neutrophil function and infectious diseases in workers occupationally exposed to phosphoorganic pesticides. *Clin. Immunol. Immunopathol.* **1984**, *33*, 13–22.
5. Thrasher, J.D.; Madison, R.; Broughton, A. Immunological abnormalities in humans exposed to chlorpyrifos: preliminary observations. *Arch. Environ. Health* **1993**, *48*, 89–93.
6. Hermanowicz, A.; Nawarska, Z.; Borys, D.; Maslankiewicz, A. The neutrophil function and infectious diseases in workers exposed to organochlorine insecticides. *Int. Arch. Occup. Environ. Health* **1982**, *50*, 329–340.
7. Kashiap, S.K. Health surveillance and biological monitoring of pesticide formulation in India. *Toxicol. Lett.* **1986**, *33*, 107–114.
8. Menconi, S.; Clark, J.M.; Langenberg, P.; Hryhorczuk, D. A preliminary study of potential human effects in private residences following chlordane applications for termite control. *Arch. Environ. Health* **1988**, *43*, 349–352.
9. McConnachie, P.R.; Zahalsky, A.C. Immune alterations in humans exposed to the termiticide technical chlordane. *Arch. Environ. Health* **1992**, *47* (4), 295–301.
10. Fiore, M.C.; Anderson, H.A.; Hong, R.; Gilubjatnikov, R.; Seiser, J.E.; Nordstrom, D.; Hanrahan, L.; Belluk, D. Chronic exposure to aldicarb-contaminated groundwater and human immune function. *Environ. Res.* **1986**, *41*, 633–645.
11. Mirkin, I.R.; Anderson, H.A.; Handaran, L.; Hong, R.; Gilubjatnikov, R.; Belluk, D. Changes in T-lymphocytes distribution associated with ingestion of aldicarb-contaminated drinking water: a follow-up study. *Environ. Res.* **1990**, *51*, 35–50.
12. Faustini, A.; Settini, L.; Pacifici, R.; Fano, V.; Zuccaro, P.; Forastiere, F. Immunological changes among farmers exposed to phenoxy herbicides: preliminary observations. *Occup. Environ. Med.* **1996**, *53*, 583–585.
13. McConnachie, P.R.; Zahlsky, A.C. Immunological consequences of exposure to pentachlorophenol. *Arch. Environ. Health* **1991**, *46* (4), 249–253.
14. Colosio, C.; Maroni, M.; Barcellini, W. Toxicological and immune findings in workers exposed to pentachlorophenol (PCP). *Arch. Environ. Health* **1993**, *48* (2), 81–88.
15. Daniel, V.; Huber, W.; Bauer, K.; Opelz, G. Impaired in-vitro lymphocyte responses in patients with elevated pentachlorophenol (PCP) blood levels. *Arch. Environ. Health* **1995**, *50* (4), 287–292.
16. Klemmer, H.W.; Wong, L.; Sato, M.M.; Reichert, E.L.; Korsak, R.J.; Rashad, M.N. Clinical finding in workers exposed to pentachlorophenol. *Arch. Environ. Contam. Toxicol.* **1980**, *9*, 715–725.
17. Colosio, C.; Tomasini, M.; Cairoli, S.; Foà, V.; Marinovich, M.; Galli, C.L. Occupational triphenyltin acetate poisoning: a case report. *Br. J. Ind. Med.* **1990**, *48*, 136–139.
18. Luster, M.I.; Rosenthal, G.J. Chemical agents and the immune response. *Environ. Health Perspect.* **1993**, *100*, 219–226.

Insect Pest Dispersal

V. J. Shivankar

Department of Entomology, National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

C. N. Rao

Shyam Singh

National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

INTRODUCTION

The basic nature of a pest population is to increase, and unless it is controlled by changing climate, heavy predation or parasitism, or artificial control measures, i.e., pesticide spraying, there is usually dispersal of part of the population to alleviate the competition pressure for food or other limited resources. Dispersal is the movement of individuals into or out of the population. It plays an important role in the geographic distribution and also in understanding the dynamics of insect pests. It may be an advantage to a species to breed individuals that rapidly seek a new place to live, although the old one might have been quite favorable. In the case of aphids and whiteflies, dispersal of only a few individuals that are insecticide resistant or virus vectors into a crop may give rise to offspring that will cause a significant impact on crop yields. Therefore, dispersal of insect pests is a topic of great concern to individuals developing various integrated pest management programs.

DEFINITION

“Dispersal” may be defined as a form of movement which leads to the removal of a variable percentage of individuals from an area to other places, irrespective of the favorableness of the latter.^[1] It is more simply defined as movement that results in an increase in the mean distance between individuals.^[2] There is a directionality to the dispersal and a minimum of turning and backtracking.^[3] Dispersal is an advantageous adaptation to countering the ephemeral availability of resources facing most insect populations and largely random movement outward from centers of high density.^[3] Other probable mechanisms of dispersal include those associated with the search for food or a mate, *phoresy*, some physiological reasons, avoiding predators, and responses to gradients of environmental factors, e.g., temperature, wind moisture, light and CO₂.^[3]

WAYS OF DISPERSAL

The dispersal of insect populations occurs in three ways: emigration, immigration and migration. Both migration and dispersal may lead to emigration of a pest from one crop and its eventual immigration into another. Local dispersal by insects may be effected by migratory behaviors, or by short host-seeking flights. The other ways in which insects can disperse include drifting with currents of air or water, by swimming, walking, flying, clinging to some moving objects including articles of commerce, etc.

Emigration involves the outward movement of an organism from one place or country to another for permanent settlement which results in depopulation. Equilibrium of population is maintained in such circumstances by enhancing the reproductive ability as well as by decreased mortality among the individuals.^[4]

Immigration involves the inward movement of the organism to any place or country. It will lead to a rise in population level, causing an overpopulation. These immigrations result in increased mortality among the immigrants or decreased reproductive capacity of the individuals.^[4]

Migration involves the mass movement of an entire population, where some insects return again to the area from which they had moved. Such movements generally take place during unfavorable conditions from the original area to other areas where conditions are favorable. Such movements are generally seasonal or periodical.^[4]

TYPES OF MIGRATION

Migration is accomplished mainly by flight and the direction of displacement for many is influenced by the wind. It may occur by ways other than flight, e.g., army ants (*Eciton hamatum*) migrate on the ground (pedestrian migration).^[3] Migration appears to be a unique phenomenon initiated by intrinsic and extrinsic factors enabling wider dispersion of

population. Three types of migration may be recognized on the basis of adult life span: 1) short-lived adults that emigrated and die within a season; 2) short-lived adults that emigrated and return; and 3) long-lived adults that hibernate or estivate.^[3] Members of the first group usually leave the breeding site, oviposit elsewhere and die, e.g., locusts, termites, aphids, thrips, and many butterflies. Relatively short-lived adults, which emigrate and return, depart from the breeding site to feeding sites, where the eggs mature. The females then fly back to the vicinity of the original breeding site and oviposit. This emigration and return may be repeated in a given season by the same individual, e.g., dragonfly species. Insects in the third category fly to hibernation or estivation sites and return to the original breeding site the following season. The monarch butterfly (*Danaus plexippus*), many noctuid moths, and several beetles fall into this category.^[3]

Migratory movements are common among locust, butterflies, aphids, and some Coleoptera and Hemiptera. The monarch butterflies (*D. plexippus*) travel very long distances and their migration is pathed every year through their conventional routes and the movement is initiated by the oncoming winter and the return trip being influenced by spring.^[3] The desert locust (*Schistocerca gregaria*) found in abundance in and around the desert areas of Africa and west Asia, which have two broods a year, show regular to-and-fro movements, one during winter and spring, while the other during summer and rainy season.^[3]

FACTORS AFFECTING DISPERSAL

Many pest species increase in numbers in zone of natural abundance (endemic), and when the population density is high some disperse into zone of occasional abundance and zone of possible abundance from time to time. The dispersal success of a pest organism depends upon several factors, including the effectiveness of the precise method of dispersal and the adaptability of the pest, particularly those of a eurythermal physiology and a polyphagous nature in relation to food.

Locomotory organs: Active means of dispersal are common in insects with well-developed powers of locomotion (by legs and wings of insects). The caterpillars of certain moths move in huge swarms over great distances and thus come to be widely distributed. Dispersal on wings is limited generally by the velocity and duration of flight of a species. Some insects like *Geotrupes* fly at a rate of 7 m/sec, *Bombus* at 3–5 m/sec, honeybees at 2.5–3.75 m/sec, and *Chrysopa perla* at 0.6 m/sec.^[5]

Wind: Light wind is an important factor in wide dispersal of delicate insects, but heavy bodied insects

like grasshoppers, locusts, beetles, etc., are also carried over great distances, across continents and oceans by strong winds. The primary means of dispersal of the Colorado potato beetle (CPB), *Leptinotarsa decemlineata*, is by wind-assisted transport within continents and oceanic crossings via human assistance.^[6] Convergent wind plays a major role in the dispersal of locust swarms. The updraft warm air currents rising from heated ground are usually strong enough to lift many low-flying insects like Coccinellids and other beetles, butterflies, dragonflies, grasshoppers, etc., to great heights in the air and carry them across plains, valleys and low hills, to high mountains and snowfields, at an elevation of 3000–4500 m on the Alps, Himalaya, and North American mountains.^[3]

Topography: The different kinds of local weather created by topographic features can influence dispersing insects at least as much as they influence the development and survival of more sedentary insect stages.

Humans: Man has both unwittingly and wittingly brought about the worldwide dispersal of a great many different species of insects. The common cockroach, bedbugs, rice weevil, granary weevil, the CPB, *Carpophilus hemipterus*, *Silvanus surinamensis*, *Lasioderma serricorne*, *Bruchus pisorum*, *B. obtectus*, *Ephestia kuehniela*, etc., are some of the common insects that have become widely distributed all over the world by human agency. Over 100 species of beetles have been introduced passively by European Settlers in North America, besides hundreds of other insects. Apart from this, man has also actively introduced a number of useful and beneficial species including parasites and predators of agricultural pests into new distant habitats.

Temperature: Temperature influences the dispersal rate of insect populations. The weevil, *Sitophilus oryzae*, concentrated with a foot or two of the surface of wheat kept for long in a large bin. As the temperature reaches and passes 32°C due to the heat of metabolism of the immobile young stages in the grain, the adults moved away to cooler places.^[7] The desert locust, *S. gregaria*, took off the mass flight between 17 and 20°C. Swarms occasionally migrated when the temperature was as low as 14–16°C and that too when the maximal temperature for the previous day had been low.^[8]

Overcrowding: Many pest species are polymorphic, containing both dispersing and sedentary forms. Crowding coupled with reduced host nutrition results in a higher population of longer-winged forms (dispersing form) of the saltmarsh planthopper (*Prokelisa marginata*) as compared to short-winged morphs (sedentary form). The movement of thrips to and from the flowers was independent of thrips density.^[9]

Host nutrition: Dispersal may be related to qualitative variation in a single population at a single time.

Most phytophagous insects feed on specific groups of plants, so their distribution is severely limited by the distribution of the food-plant. Bimodal immigration of aphids toward collard plants in response to the decline in quality of nearby *Brassica* spp. as suitable hosts for cabbage aphids has been reported.^[2]

Deserts and oceans: Extremes of aridity/humidity/salinity restrict the diversity and density of insects in desert/ocean regions, where they also contend with high/low temperatures, loose inorganic soil surfaces, and wind. Escape in time and space is achieved in many cases by shifts in diurnal and seasonal rhythms, but the extremes can often be tolerated only because of specialized morphology.

FUTURE PROSPECTS

Dispersal is the most important component of insect population processes, yet, in general, spatial dynamics of insect populations are not as well studied (or as well modeled) as are temporal dynamics.^[3] Probably the most important single line of research required in population ecology is to find better ways of quantifying the effects of dispersal.^[10] Dispersal is often omitted as a component of IPM programs because too little is known about the factors that influence migration and dispersal by a particular insect pest and testing the impact of pest dispersal phenomenon on pest populations in agricultural settings is extremely difficult. The interactions between migration and other aspects of pest biology are also critical in developing IPM strategies to deal with mobile pests. The knowledge of insect migration and dispersal would lead to the development

of simulation models that serve as an aid to growers in making management decisions.^[10]

REFERENCES

1. Clark, L.R.; Geier, P.W.; Hughes, R.D.; Morris, R.F. *The Ecology of Insect Populations in Theory and Practice*; Methuen and Co. Ltd.: London, 1967.
2. Horn, D.J. *Ecological Approach to Pest Management*; The Guilford Press, A Division of Guilford Publications, Inc.: New York, 1988; 285 pp.
3. Romoser, W.S.; Stoffolanoz, J.G., Jr. *The Science of Entomology*, 3rd Ed.; C. Brown Publishers: USA, 1994; 532 pp.
4. Yazdani, S.S.; Agarwal, M.L. *Elements of Insect Ecology*; Narosa Publishing House: New Delhi, 1997; 209 pp.
5. Mani, M.S. *General Entomology*; Oxford & IBH Publishing Co. Pvt. Ltd.: New Delhi, 1982; 912 pp.
6. Hare, J.D. Ecology and management of the Colorado potato beetle. *Annu. Rev. Entomol.* **1990**, *35*, 81–100.
7. Birch, L.C. The movement of *Calandra oryzae* L. (small strain) in experimental bulks of wheat. *J. Aust. Inst. Agric. Sci.* **1946**, *12*, 21–26.
8. Gunn, D.L.; Perry, F.C.; Seymour, W.G.; Telford, T.M.; Wright, E.M.; Yeo, D. Behaviour of desert locust (*Schistocerca gregaria* Forsk) in Kenya in relation to air craft spraying. *Bull. Anti-Locust Res. Centre*, London **1948**, *3*, 138 pp.
9. Denno, R.F. Tracking variable host plants in space and time. In *Variable Plants and Herbivores in Natural and Managed Systems*; Denno, R.F., McClure, M.S., Eds.; Academic Press: New York, 1983; 291–341.
10. <http://phylogeny.arizona.edu/ENTO/entohome.html> (accessed August 2001).

Insect Pest Management

Thomas J. Henneberry

*Arid Land Agricultural Research Center, USDA-ARS,
Maricopa, Arizona, U.S.A.*

INTRODUCTION

Worldwide, current farm values of crop and animal production are estimated at more than \$1.3 trillion.^[1] Various authors, as reviewed by Schwartz and Klassen,^[2] have suggested farm production losses by arthropod pests that appear to be in the range of 10% to 15% with additional losses of 10% to 40% occurring during post-harvest handling. The cost of pest control in the United States and worldwide is estimated to be over \$20 and \$120 billion, respectively.^[3] Efforts to reduce these losses and control costs have been a driving force in agricultural research. Over the past two decades, increases in world food production have exceeded population growth in most countries. Since the 1960s, worldwide agricultural production has increased 80%. However, a continuation of this trend is not assured.

The world's human population exceeds 6 billion people.^[4] If the population growth rate is only 1%, an additional 165,000 people are added daily. These and similar demographics have intrigued and challenged scientists to develop new food and fiber production technology to provide for the needs of escalating human population growth. The competitive struggles between man and arthropod pests for the products of man's agricultural labors have existed since the beginning of time. The revolutionary discovery of DDT and subsequently thousands of other synthetic organic chemicals for insect control placed insecticides in the forefront of insect control methodology. Their impact on reducing arthropod-borne diseases and achieving high crop and animal productivity has been unparalleled. However, these advances were not made without cost. Heavy reliance, misuse, and overuse of insecticides, in some instances, posed a threat to human health and resulted in development of insect resistance, environmental contamination, adverse effect on non-target organisms, and development of secondary pests. When the bright future of the insecticide era became clouded with these issues, research, regulatory and extension activities were challenged to maintain or increase crop and animal production, within the context of more ecologically acceptable pest control methodology. Foremost among the advanced concepts to provide economically,

environmentally, and socially acceptable insect control was integrated pest management (IPM). The concept originally addressed insect pest management but was broadened to include disease, weeds, and other pests. The origin of the terminology can be traced^[5] from integrated control,^[6] which became synonymous with integrated pest management^[7] and pest management.^[8] The Entomological Society of America^[9] defined IPM as: "A pest management system that in the context of the associated environment and the population dynamics of the pest species utilizes all suitable techniques and methods in as compatible a manner as possible and maintains the pest populations at levels below those causing economic injury."

INSECT PEST MANAGEMENT

Integrated pest management approaches to insect suppression with the goals of reducing crop and animal losses in quality and yield and increase net profits to the producer provide exciting challenges. Ideally, control methods cause minimal environmental damage and pose little or no risk to human health. The focus on large areas that include as much of the total target pest population as possible evolved with our increasing awareness of the limitations of attacking local infestations.^[10] Area-wide approaches involve the coordinated cooperative efforts of all parts of agricultural communities. Integrated pest management systems combine methods such as chemical control, crop rotation, crop sanitation, time of planting, host-free periods, resistant varieties, and genetic and biological control into a single pest control effort. Chemical control action is based on need and is determined using economic injury and action threshold decision-making tools. Multiple pest suppression techniques integrated into a single management system have the highest probability of successful long-term crop protection from insect pest.

The essentials for successful IPM programs include knowledge of 1) crop and animal production methods; 2) biology and ecology of each pest species; 3) basic information on genetics, behavior, and physiology of pest species; 4) relationships and interactions of the pests with the crop and other biological and physical

components of the ecosystem; and 5) potential economic damage of each pest complex.

Control methods must be compatible with crop production methods and the ecosystem. Cultivar selection and planting date, as well as cultural practices (irrigation, fertilization, and tillage), may have a major influence on pest severity. Decisions on the need for control action are based not only on these factors but also on pest population levels, the present and predicted weather, the levels of existing biological control, and the stage of plant development and potential for yield losses.

TECHNOLOGIES AND PROCEDURES

Economic Thresholds

The economic threshold is the population level below which the cost of taking control action exceeds the losses caused by the pest. Pest populations that can be tolerated within a crop system can vary because of crop harvesting schedules and inherent crop tolerance to pest attack. These thresholds may also vary from area to area and among farms that are in the same area but under different management systems. They may need to be adjusted when two or more pests are attacking the same crop. Using economic thresholds to determine the need for control action has helped reduce the number of insecticide applications, increased grower net profits, and reduced insecticide resistance development.

Sampling Technologies

Cost-effective sampling methods for each pest are necessary to determine pest numbers for purposes of establishing action thresholds as decision-making tools. These methods range from simple to complex and include such simple techniques as direct insect counts and damage ratings to computer imaging and geographical information systems.

Environmental Controls

The basic framework of insect pest management includes natural enemies, weather, climate, and food resources. Parasites, predators, and microbial agents are the major natural control agents that help regulate insect populations. Conservation of these natural enemies to interact with arthropod pests is a primary focus of insect pest management. Selective pesticides that are least harmful to natural enemies, microbial controls, and other non-chemical methods are encouraged.

Effects of Pest Migration and Movement

Many insects and their natural enemies disperse as their populations grow. The reasons include crowding, search for food as a result of host depletion, or passive movement by winds and atmospheric weather patterns, and/or they may be transported unintentionally in or on plant products. Migration and dispersal patterns are of particular importance in area-wide management systems. Effective natural barriers such as mountains or large bodies of water may be exploited. Artificial barriers such as the release of sterile insects or quarantine of certain plants and produce can be useful to prevent or reduce unwanted movement of pests.

Host Plant Resistance

Many plants have evolved resistance mechanisms that enable them to prevent or survive insect attacks. Geneticists have made outstanding progress in identifying and incorporating these pest resistance characteristics in commercial cultivars. Much progress has been made in finding genes for resistance and transferring them to plants. The rapid development of new methods for gene transfer promises that host resistance will play a much greater role in IPM in the future.

Insecticides

Synthetic pesticides have been a major factor in farmers' ability to cope with insect pests. This will probably continue to be the case in the future. Resistance development, undesirable environmental, and social effects are major issues. The judicious use of insecticides on a need basis, new chemistry, and insect resistance management techniques are reducing the undesirable side effects of insecticides.

Modeling

From the foregoing, it is obvious that farm systems are complex. Changes in one operation affect others. Economic factors and social pressures as well as biological systems are involved in decision making. Models help us understand the complexities of biological systems, improve decision-making at the farm level, and most importantly, models require the user to define available knowledge and provide information to explain deficiencies that result in differences between model results and field observations.

Implementing Integrated Pest Management

The implementation of insect management systems requires extensive research, extension and technology transfer, farmer time, and community effort. Often, significant modifications in farming practices must be made. Changes might include crop rotation, destruction of crop residues, and variations in time of planting. Management systems can be adopted by individual farmers, by small groups, or by farmers across broad agricultural systems. The insect problem may dictate whether a single, small group, or regional adoption will be most effective. Where farms are scattered, crop diversity and insect migration from other farms may not be a factor; adoption by an individual may be appropriate. In specialized areas with extensive monoculture or crops where pests move freely from one farm to another, all farmers must participate for successful implementation. The approach focuses on the total insect population, as opposed to efforts by individual farms or small local areas attempt to control limited segments of the insect population. Area-wide programs include researchers, producers, extension personnel, and private consultants as active participants in the program. The entire community has a part in the program.

Success Stories

Successful programs have provided economic benefits to farmers and more environmentally acceptable crop protection practices. Some of the outstanding successes, such as the boll weevil eradication program, have relied on early detection, selective insecticide use, and cultural practices. Mediterranean fruit fly, pink bollworm, and screwworm programs have used sterile insect releases as the main suppression component supported by intensive population sampling, attractants, and cultural practices. The highly successful codling moth area-wide program uses mating inhibition with sex pheromone as its main IPM component, whereas the foundation of effective alfalfa aphid management is host plant resistance. A complex of imported parasite species has been used to manage alfalfa weevil populations. These and many other examples are exciting evidence of practical applications of IPM concepts and provide a glimpse into the future of socially, environmentally, and economically sound pest control.

CONCLUSION

IPM technology is dynamic and improving with trial experience, implementation, and acceptance. Our increasing knowledge and information retrieval capabilities provide new insights into the potential of innovative pest management. New crop protection technologies and safer, more environmentally compatible pesticides have greatly expanded the arsenal from which effective IPM programs can be constructed. Existing and past IPM programs provide good examples where biological, chemical, behavioral, and cultural controls have melded together with host plant resistance and transgenic crops to produce stable and effective pest suppression. There is much opportunity to build on past successes and much optimism for the future of ecologically oriented pest management.

REFERENCES

1. <http://www.fao.org/WAICENT/FAOINFO/ECONOMIC/ESS/2000/totalpro/VARIOUSG.GIF> (accessed October 2000).
2. Schwartz, P.H.; Klassen, W. Estimate of losses caused by insects and mites to agricultural crops. In *Handbook of Pest Management in Agriculture*; Hanson, A.A., Pimental, D., Eds.; V.R.I. CRC Press, Inc.: Boca Raton, FL, 1981; Vol. 1, 15–77.
3. Pimental, D. New York State College of Agriculture and Life Sciences, Cornell University: Ithaca, NY. Personal Communication.
4. <http://www.census.gov/cgi-bin/ipc/popclockw> (accessed October 2000).
5. Bottrell, D.R. *Integrated Pest Management*; Council Environ. Qual., U.S. Govt. Printing Office: Washington, DC, 1979; 120 pp.
6. Stern, V.M.; Smith, R.F.; Hagen, R.S. The integrated control concept. *Hilgardia* **1959**, *29*, 81–101.
7. Smith, R.F. History and complexity of integrated pest management. In *Pest Control Strategies*; Smith, E.H., Pimental, D., Eds.; Academic Press: New York, NY, 1978; 41–53.
8. Geier, P.W.; Clark, L.R. *An Ecological Approach to Pest Control*, Proc. 8th Tech. Mtg. Int'l. Union Conserv. of Nature and Natural Res., Warsaw, 1961; 10–18.
9. Glass, E.H.; Murdoch, W.P.; Packer, J.S. Integrated pest management: rationale. Potential, needs and improvement. *Spec. Publ. Entomol. Soc. Am.* **1975**, *75–82*, 141.
10. Knipling, E.F. The basic principles of insect population suppression and management. In *USDA Agricultural Handbook*; U.S. Department of Agriculture: Washington, DC, 1979; 659 pp.

Insect Pest Management: Lawns

Frederick P. Baxendale

Department of Entomology, University of Nebraska, Lincoln, Nebraska, U.S.A.

INTRODUCTION

Effective and environmentally responsible insect management is an important consideration in the overall care of turfgrasses. Lawn care professionals and other turfgrass managers must not only accurately diagnose emerging insect problems, but also anticipate future pest activity (Fig. 1). This chapter presents an integrated approach to the management of insect and mite pests affecting lawns and other turf areas.

IMPLEMENTING AN INTEGRATED PEST MANAGEMENT (IPM) PROGRAM FOR TURFGRASS

Establishing an effective management program for turfgrass insects requires a sound understanding of the growth habits and cultural requirements of turfgrasses; knowledge of the biology, behavior, life history, and type of damage caused by potential pests; and information regarding the time of year, growth stage of turfgrasses, and environmental conditions under which pest activity and damage are most likely to occur. Accurate pest identification is also important. In addition, the turfgrass manager must integrate insect control with disease, weed, and cultural management strategies.

Pest Identification

All turfgrasses are inhabited by a diverse array of organisms including insects, spiders, mites, nematodes, and many other small animals. Most cause little or no damage and are generally considered non-pests. Others serve important beneficial roles in the breakdown of thatch, aeration of the soil, or as natural enemies of various insect and mite pests. Only a few of the species present are actually plant-feeding pests (Fig. 2). Because of the many similarities between pests and non-pests, it is essential that the turfgrass manager accurately distinguish incidental and beneficial species from target pests.

Early Detection

Successful management of most turf insects depends on the early detection of pests before they reach

damaging levels. This can best be accomplished through frequent turf inspections to detect early signs of insects and mites, and their damage. Among the more common symptoms of insect-damaged turf are general thinning of the grass, spongy areas, irregular brown patches, and/or plants that easily break away at soil level (Fig. 3). However, confirming the insect origin of the problem can be difficult because many of the symptoms described above could also have been caused by non-insect factors such as heat or drought stress; nutritional deficiencies; turf diseases; soil compaction; chemical burns from gasoline, fertilizers, herbicides, or insecticides; scalping during mowing operations; or even excrement spots left by pets. If the problem is insect-related, a close visual inspection of the damaged area should reveal either the presence of the pest, or indirect evidence that insect infestation has been present.

Bird and animal feeding activity often indicates a potential insect problem (Fig. 4). Flocks of foraging birds, particularly starlings and robins, and/or digging and tunneling by skunks, raccoons, armadillos, moles, or other animals are common, early indicators of insect infestations. Other signs that can indicate an existing infestation or signal the potential for future problems include the presence of large numbers of scarab beetles (e.g., Japanese beetles, European and masked chafers, asiatic garden, and oriental beetles), armyworm or cutworm moths around lights, billbug adults on sidewalks and driveways, or sod webworm moths flying over lawns in the process of depositing their eggs.

Confirmation of the insect origin of the problem requires close examination of the injured area. Look for signs of skeletonized or discolored leaves, clipped grass blades, fecal pellets, sawdust-like debris, stem tunneling, silken tubes, or webbing (Fig. 5). If no insects or evidence of feeding are found, the condition is likely because of some other cause, and use of an insecticide would be of no value.

Insect Monitoring Techniques

All turf areas should be regularly inspected for pest problems throughout the growing season. Monitoring allows the turfgrass manager to confirm the presence or absence of insect or mite pests, determine the pest species present, assess the need for taking corrective



Fig. 1 Home lawn—Baxendales. (Courtesy of Department of Entomology, University of Nebraska.)

measures, evaluate the efficacy of insecticide treatments, and develop site history information.

Insect monitoring techniques include visual observation, soil sampling, use of irritants (e.g., detergents, Detect-Aid), pitfall traps, flotation devices, and sweep nets. Light and pheromone traps can also be used to monitor the seasonal occurrence of insects, and as indicators of when to start sampling for specific pests

(Fig. 6). Because most insect and mite pests infesting turf do not distribute themselves evenly throughout the stand, it is essential that the turf area be sampled in a consistent, uniform pattern. Enough samples should be taken to assure a reasonably accurate estimate of pest numbers in the sampled area. If turf damage is evident but no pests are detected, examine the turf for other causes of injury such as disease, excessive thatch, improper mowing, heat, or moisture stress. When examining turf, be on the lookout for beneficial natural enemies, such as ants, big-eyed bugs, ground beetles, lacewings, lady beetles, spiders, and parasitic wasps that may be reducing pest populations (Figs. 1–7).

Surface-active insects often can be detected by applying 1/4 cup of lemon-scented household detergent, or one tablespoon of 1% pyrethrins in 2 gal of water poured over one square yard of turf. These preparations irritate mole crickets, webworms, cutworms, billbug adults, and other surface-feeding pests, causing them to move to the surface in 5–10 min, where they can be counted.

For soil-active insects such as white grubs and billbug larvae, activity can be verified by cutting 1/4 ft² (6 × 6-in.) sections of turf on three sides, peeling back the sod and examining the upper 2 in. of root zone for the presence of pests. Turfgrass managers with access

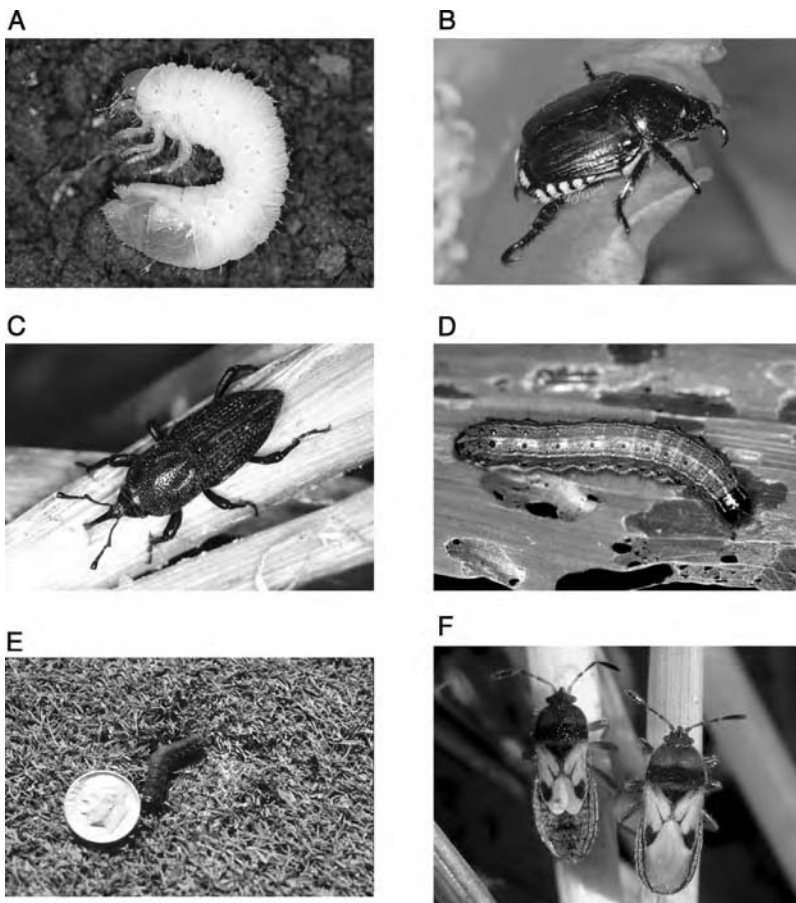


Fig. 2 (A) Lawn pest—annual white. (B) Lawn pest—Japanese beetle. (C) Lawn pest—billbug. (D) Lawn pest—fall armyworm. (E) Lawn pest—Black cutworm. (F) Lawn pest—hairy chinch bugs. (Courtesy of Department of Entomology, University of Nebraska.)



Fig. 3 Insect (WG)-damaged lawn. (Courtesy of Department of Entomology, University of Nebraska.)

to a golf course cup cutter also can sample for soil-inhabiting insects by taking 4-in.-diameter (0.1 ft²) turf soil cores.

Recordkeeping

Accurate recordkeeping is essential for the success of a turfgrass pest management program. Records should be as complete as possible and include the kinds and numbers of pests present, when and where they were found, and exact locations and extent of any turf damage or abnormalities observed. Information on the turf species and cultivar development, turf health, and current environmental conditions is also valuable. At the end of the season, review this information and make plans to improve your pest management program for next year. You may have detected certain patterns, such as a greater number of pests or more damage in some areas or associated with certain cultivars, which will help you focus future monitoring and management activities.

A



PEST MANAGEMENT ALTERNATIVES

IPM uses a combination of complementary strategies to effectively manage pest populations. The following paragraphs describe some of the pest management alternatives available to the turfgrass manager.

Cultural Methods

Turfgrass selection

Select turfgrass species or cultivars that are well adapted to local soil and environmental conditions. Adapted turfgrasses are better able to tolerate stress, and are less likely to be damaged by insects than poorly adapted grasses. Furthermore, a blend of improved adapted grasses will usually outperform a single cultivar. Information on locally adapted turfgrasses is available from your local turfgrass specialist, cooperative extension office, as well as most nurseries and garden centers.

Effective turfgrass management

Many insect pests that infest turfgrasses are attracted to lush, overly maintained turf. Sound cultural practices that optimize plant health and vigor enable the turf to withstand higher pest infestation levels and recover more rapidly from insect and mite injury. Careful turfgrass management is one of the best insect prevention strategies available.

Insect-resistant and endophyte-enhanced grasses

Planting insect-resistant turfgrasses is another valuable IPM tool. Plant resistance to insect pests has been found in many plants, although the degree of resistance may vary considerably from one species or cultivar to

B



Fig. 4 (A) Bird and animal damage. (B) Starling. (Courtesy of Department of Entomology, University of Nebraska.)

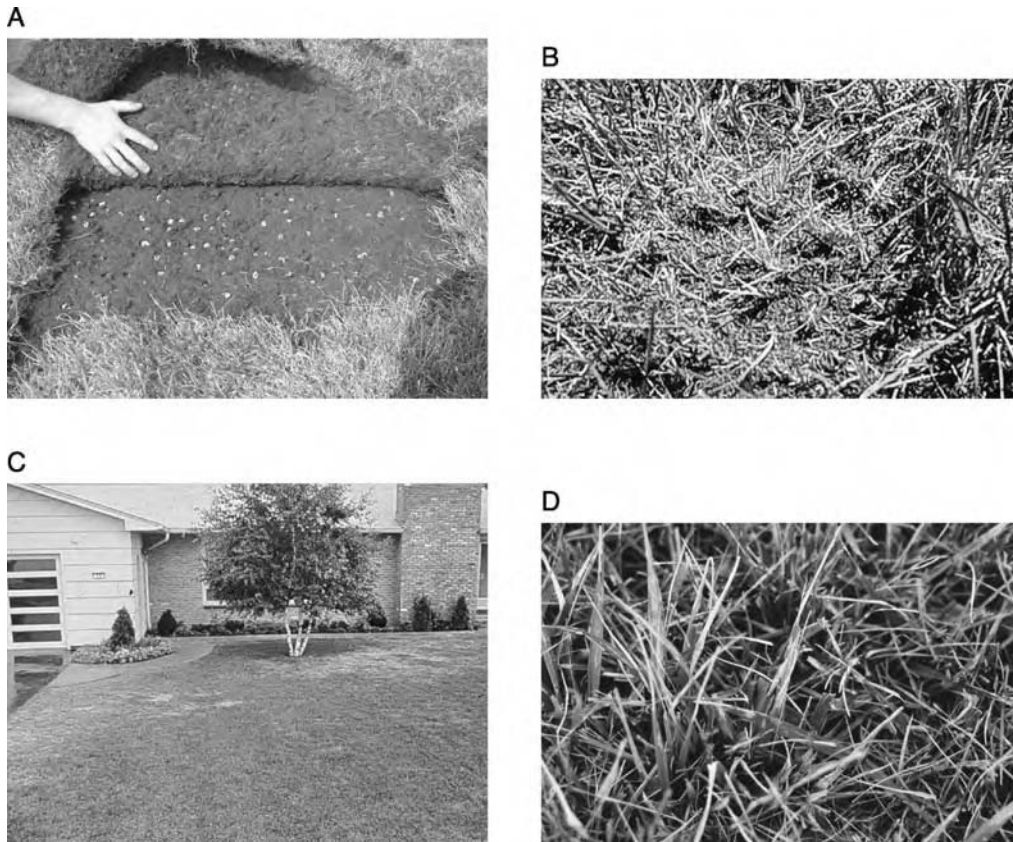


Fig. 5 (A) White grubs and damage. (B) Sod webworm damage. (C) Billbug damage. (D) Fall armyworm damage. (Courtesy of Department of Entomology, University of Nebraska.)

another. Several cultivars of billbug-resistant Kentucky bluegrass are commercially available.

Endophyte-enhanced grasses have also shown resistance to numerous turfgrass insect pests including aphids, leafhoppers, chinch bugs, armyworms, webworms, and billbugs. Among the turfgrasses containing endophytes are cultivars of perennial rye, tall and fine

fescues. Unfortunately, useful endophytes have not been found in creeping bentgrass or Kentucky bluegrass.

Biological Control

This important IPM strategy utilizes beneficial organisms including predators, parasitoids, or insect

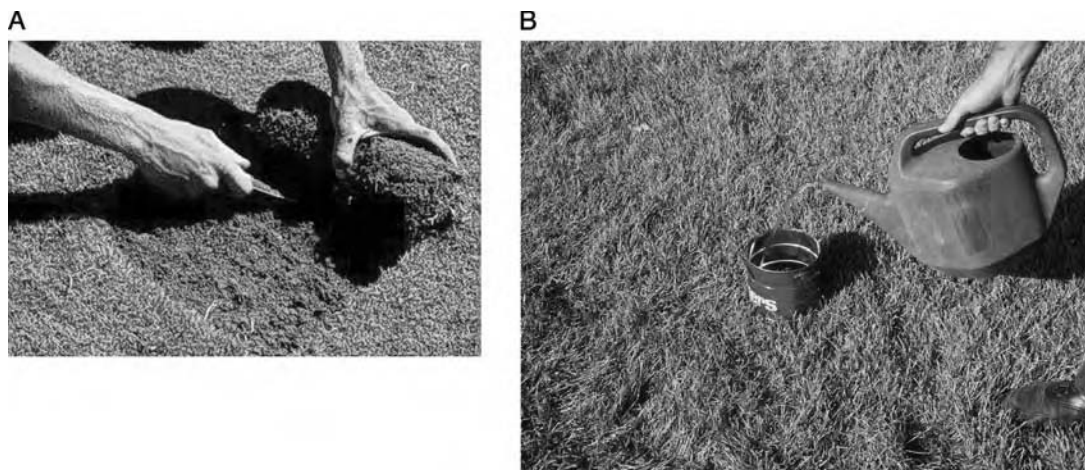


Fig. 6 (A) Sampling for white grubs. (B) Flotation. (Courtesy of Department of Entomology, University of Nebraska.)

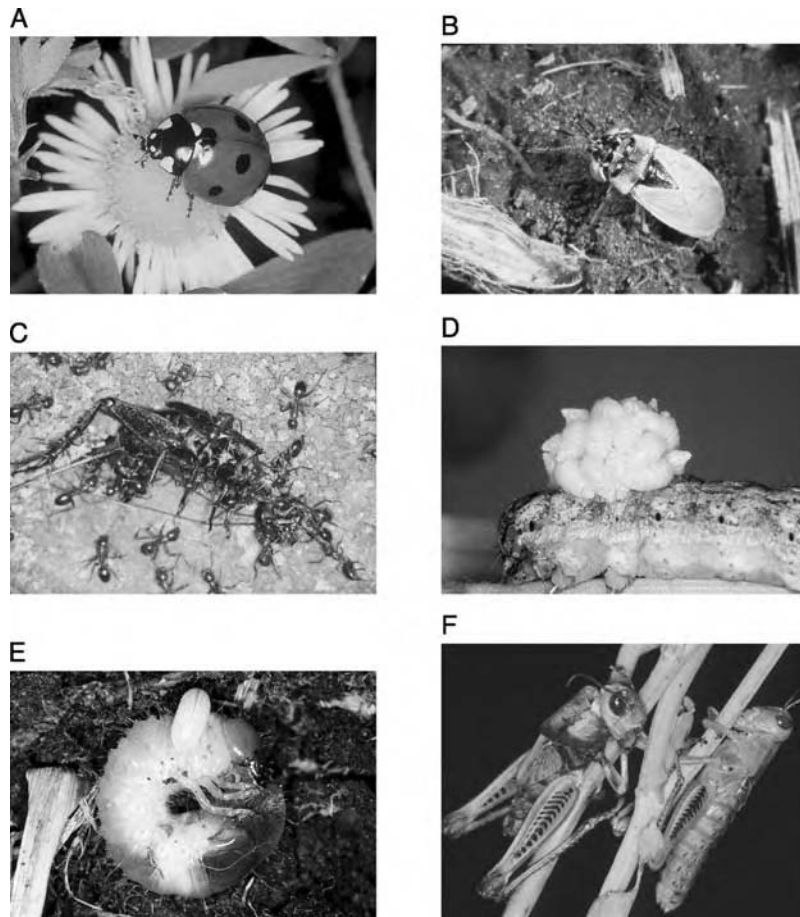


Fig. 7 (A) Ladybeetle. (B) Big-eyed bug. (C) Ants attacking a cricket. (D) Eulophids on armyworm. (E) White grub with parasitoid. (F) GH with *Nosema*. (Courtesy of Department of Entomology, University of Nebraska.)

pathogens to reduce pest populations. In general, effective use of this approach requires a detailed knowledge of predator/prey or parasitoid/host biology, accurate timing, and careful application procedures.

Beneficial insects and mites

Natural populations of predators (e.g., ants, big-eyed bugs, ground beetles, lacewings, lady beetles, predaceous thrips, and mites) and parasitoids (e.g., parasitoid wasps, tachinid flies) are valuable in reducing infestations of insect and mite pests (Fig. 7). If these or other beneficial organisms are observed in the turf, care should be taken to ensure their survival. If pest control becomes necessary, corrective measures that minimize injury to beneficial organisms should be selected. Remember, low pest infestation levels may need to be tolerated to attract and maintain natural enemy populations.

Disease-causing microorganisms

Certain insect pathogens (disease-causing organisms) or their products can also be used to reduce insect infestations. Among the microorganisms known to

infect turfgrass insects are bacteria, fungi, viruses, protozoans, and nematodes. Products containing many of these insect pathogens are available through pest management supply companies and some pesticide manufacturers.

Insecticides/Acaricides

Insecticides and acaricides are the most powerful tools available for insect and mite control in turf. In many cases, they afford the only practical method of reducing pest infestations that have already reached damaging levels. Insecticides have rapid corrective action and offer a wide range of properties and methods of application. They are relatively low in cost, and their use often results in a substantial economic or aesthetic benefit. Among the potential problems associated with insecticide use are development of pest resistance; outbreaks of secondary pests; adverse effects on non-target organisms including humans, pets, wildlife, and beneficial insects; hazardous residues in our food supply; and ground water contamination.

When insecticides are used in an IPM program, careful product selection and timing of applications

are extremely important in obtaining the best possible pest control with the least adverse effect on the environment. Observe aesthetic/damage threshold levels (i.e., treat only when necessary) and, wherever possible, limit applications to infested areas of the turf. Ensure proper calibration of the application equipment and always read, understand, and follow label directions.

CONCLUSION

Establishing an IPM program for lawns and other turf areas will require time, effort and careful planning. However, the potential rewards are substantial in terms of improved insect and mite control, cost savings, and reduced reliance on pesticides.

For additional information on turfgrass insects and their management, refer to the following sources.

BIBLIOGRAPHY

- Fermanian, T.W.; Shurtleff, M.C.; Randell, R.; Wilkinson, H.T.; Nixon, P.L. *Controlling Turfgrass Pests*, 2nd Ed.; Prentice-Hall: Upper Saddle River, NJ, 1997.
- Brandenburg, R.L., Villani, M.G., Eds.; *Handbook of Turfgrass Insect Pests*; Entomological Society of America: Hyattsville, MD, 1995.
- Baxendale, F.P., Gaussoin, R.E., Eds.; *Integrated Turfgrass Management for the Northern Great Plains, EC97-1557*; University of Nebraska Cooperative Extension: Lincoln, NE, 1997.
- Niemczyk, H.D.; Shetlar, D.J. *Destructive Turf Insects*, 2nd Ed.; HDN Books: Wooster, OH, 2000.
- Potter, D.A. *Destructive Turfgrass Insects: Biology, Diagnosis and Control*; Ann Arbor Press: Chelsea, MI, 1998.
- Villani, M.G.; Vittum, P.J.; Tashiro, H. *Turfgrass Insects of the U.S. and Canada*, 2nd Ed.; Cornell University Press: Ithaca, NY, 1999.

Insecticide Reduction on Lawns

Eileen A. Buss
Philip Koehler

*Entomology and Nematology Department, University of Florida,
Gainesville, Florida, U.S.A.*

INTRODUCTION

Turfgrasses are environmentally important in urban areas. They reduce water and soil erosion, filter synthetic organic compounds, trap and filter storm water runoff, and provide flood control. Healthy lawns improve the aesthetics of the landscape and increase property value. Turf on sports fields, golf courses, parks, and other recreational areas contribute to people's overall health and quality of living. Maintained turfgrass areas are critical for visibility, security, and safety on roadways, airfields, and other sensitive areas.^[1] Most of the turfgrass acreage in U.S.A. is devoted to home lawns.

Many arthropods live or feed in lawns, including foliage feeders (e.g., armyworms, cutworms, and sod webworms), sap feeders (e.g., aphids, chinch bugs, leafhoppers, mealybugs, mites, scale insects, and spittlebugs), stem borers (e.g., billbugs), root feeders (e.g., flies, ground pearls, mole crickets, and white grubs), and beneficial organisms (e.g., parasitoids, predators, pathogens, pollinators, and decomposers).^[2] The importance of each pest group may vary by time of year, geographic location, or the turfgrass species that is infested. In general, white grubs are considered the most damaging insects in cool-season turfgrasses, and mole crickets are the most damaging to warm-season turfgrasses. The other pests may be more sporadic.

In pest management, identification of a symptom or pest is just the first step. Understanding why the symptom occurs, and modifying the way the turfgrass is grown is the real challenge for sustainable control. A "reactive" person may treat symptoms of pest problems without determining the various factors that contributed to the outbreak. A "responsive" person calls upon various resources (e.g., experience, training, test results, references, or experts) to determine which factors worked together to cause the problem, and then tries to modify the system to reduce the chance of it occurring again. Insecticides are used selectively. Many pest problems can be prevented or minimized by properly maintaining healthy turfgrass.^[3] Pest outbreaks tend to occur when turfgrass is too stressed to outgrow damage or when adults are attracted to a site and

preferentially oviposit in that location. Resistance of different turfgrass species and cultivars to insect herbivory is likely related to their ability to tolerate damage.^[4]

FERTILIZATION

Fertilization is perhaps the most important cultural practice that affects turfgrass insect pests. Organic fertilizers such as turkey or chicken litter that are applied to turfgrass can attract green June beetle adults during the summer and result in significant grub damage.^[5] Overfertilization of turfgrass attracts fall armyworm and grass looper moths, which results in succulent leaf tissues and rapid buildup of caterpillar populations.^[6,7] An overfertilized lawn may have a thicker thatch layer, which provides ample habitat to thatch-dwelling pests such as chinch bugs and spittlebugs. Sometimes stresses such as overfertilization and mowing can also reduce host plant resistance and allow insects to feed and survive on them.^[8] Lawns that receive the proper source and amount of fertilizer for their growing conditions are more likely to have a dense enough canopy to prevent weed encroachment and may tolerate some feeding injury.

IRRIGATION

The amount of irrigation used can either positively or negatively influence turfgrass health in relation to insect-feeding damage. Adequate soil moisture is necessary for the eggs and immatures of many root-feeding insects (e.g., scarab beetles and mole crickets) to survive.^[9,10] Adults may be attracted to and lay more eggs in irrigated turfgrass during hot, dry weather. However, turfgrass that receives adequate water may have deeper root systems and be better able to tolerate or outgrow some root-feeding damage than drought-stressed turfgrass. Increased irrigation may dislodge pests from plants, insects may drown, and beneficial pathogens may spread to help suppress pest populations.^[2]

Table 1 Some key turfgrass pests, their host plants, symptoms, and suggested damage thresholds^a

| Arthropod pests | Preferred hosts | Damage symptoms | Suggested damage thresholds |
|---|--|---|-----------------------------|
| Billbugs (Bluegrass, Denver, Hunting, Phoenician) | Cool-season grasses, bermudagrass, zoysiagrass | Larvae burrow down grass stems to the plant crown, killing stems, and larger turf areas. Often misdiagnosed as drought, other insects, or disease | 7–10 billbugs/sq. ft. |
| Caterpillars | Many grasses, small grains, legumes | Skeletonized, notched, or completely consumed foliage, with bare spots | 3–8 larvae/sq. ft. |
| Chinch bugs (Hairy, Southern, Common) | Cool-season grasses, St. Augustinegrass | Foliage yellows, wilts, and dies in small spots, then larger patches. | 15–25 chinch bugs/sq. ft. |
| Mole crickets (<i>Scapteriscus</i> spp.) | Bermudagrass, bahiagrass, other warm-season grasses | Tunneling below the soil surface and root feeding result in bare patches of turf | 2–4 tunnels/sq. ft. |
| <i>White grubs</i> | | | |
| Black turfgrass atenioides | Annual bluegrass, Kentucky bluegrass, bent grasses | Root feeding, resulting in wilting and gradual thinning of turf | 40–100 grubs/sq. ft. |
| Green June beetles | Kentucky bluegrass, tall fescue, bermudagrass, thin-skinned fruits | Root feeding results in wilting and dying grass. Grubs make mounds | 5–7 grubs/sq. ft. |
| Japanese beetle | Most grasses | Grubs feed on roots and root hairs, resulting in turf wilting, and thinning Adults skeletonize tree and shrub leaves | 10–20 grubs/sq. ft. |
| Masked chafers | Pasture grasses and turfgrasses | Larval root feeding weakens grass, resulting in wilting and dieback Adults do not feed | 10–20 grubs/sq. ft. |
| May and June beetles | Many grasses | Grubs feed on roots, resulting in wilting and dieback. Adults eat leaves of grasses, herbs, shrubs, and trees | 3–6 grubs/sq. ft. |
| Oriental beetle | Turfgrasses and sugarcane | Grubs feed on roots near the soil surface Adults feed on several flowering plants | 6–8 grubs/sq. ft. |

^aThresholds vary depending on the condition and use of the turf.

MECHANICAL CONTROL

Mechanical controls such as mowing or verticutting can physically kill insect pests or modify their habitat. It is important to mow at the correct height for a particular turfgrass species and remove only one-third of the grass blades at a time. If turfgrass is mowed too low (i.e., scalped), too much leaf material is removed, the crown is damaged, photosynthesis is reduced, the root system is reduced, and turfgrass is weakened. Catching and removing the clippings after mowing can reduce populations of cutworms if eggs are laid on the grass blades.^[11] Even raising mowing heights may decrease insect survival while increasing root mass.^[9] Verticutting can reduce thatch thickness in lawns, but temporarily hurts the lawn's appearance. Thatch reduction decreases the amount of habitat and humidity available to chinch bugs and spittlebugs.^[12,13]

It also improves the efficacy of some insecticides, especially pyrethroids, because less organic matter is present for the insecticides to bind with, and they can better penetrate the soil to contact the target pest.

BIOLOGICAL CONTROL

Insect pest populations in turfgrass may also be suppressed by natural enemies. Generalist predators such as ants, big-eyed bugs, earwigs, ground beetles, minute pirate bugs, rove beetles, and spiders are frequently present. Diverse parasitoids, pathogens (e.g., *Bacillus* spp., *Beauveria bassiana*, *Metarhizium anisopliae*, *Paenibacillus* spp., and *Pasteuria* spp.), and insect parasitic nematodes (e.g., *Heterorhabditis* spp. and *Steinernema* spp.) may also be naturally present, and some may be purchased and released into turfgrass.^[2]

Exactly how useful these natural enemies are has not been determined for every pest species, but the effects of synthetic insecticide applications to many of the beneficials have been documented.^[14] Natural enemies may be conserved by using spot treatments rather than treating entire lawns, thus providing untreated refuge and a continued food source. Unfortunately, misidentifications can be made, and sometimes people target insecticide treatments directly against beneficials. In such cases, knowing what is a pest and what is not is critical, and is a key concept in monitoring.

CHEMICAL CONTROL

When to use an insecticide against a lawn pest is not always an easy decision. In reality, many lawns are often treated on a calendar basis, regardless of pest presence. The image or desire is to prevent any pests from becoming established through regular (e.g., quarterly) insecticide applications. But within an integrated pest management strategy, the need to treat is based on monitoring and thresholds of pest abundance, the presence of susceptible life stages, and damage potential. The general thresholds for several pests are provided in Table 1, but thresholds have not yet been determined for all potential insect pests of turfgrass. Monitoring pests can be accomplished with some knowledge of their life cycle and using several proven techniques.^[2] For instance, soapy water (1 oz of dishwashing soap per gallon of water) can be used to flush insects hidden in the upper inches of soil or in thatch. Sod can be cut and rolled back to determine numbers of grubs that are feeding on the roots. Caterpillars can be located by looking for notches in leaves, ragged patches of turfgrass, or bare areas of lawns. The highest densities of chinch bugs are usually found in green areas next to dead and dying grass. Periodic monitoring helps a homeowner find infestations before significant damage has occurred and when insects may be younger and easier to control with “softer” products such as microbials or insect growth regulators, rather than broad-spectrum synthetic insecticides.

Recent trends in regulation and public sentiment have resulted in the loss of several broad-spectrum insecticides (e.g., organophosphates such as Dursban and Diazinon and organochlorines such as Lindane) in urban areas. Insecticides that are extremely toxic to mammals are not available for use in this market niche anymore. Some insecticide application rates have also been reduced to minimize human exposure, making the products ineffective; people stop purchasing these products and they are lost from the marketplace. Even the image that a particular insecticide is toxic or “bad” is sufficient to ruin its reputation and market

potential. The remaining products tend to be more selective (kill fewer insect species), have new modes of action (molting or feeding inhibitors, insect growth regulators, or nerve toxins that act on different nervous system sites), less mammalian toxicity, break down faster (weeks or months instead of years), and be sometimes more expensive. Less active ingredient is used, which means that less insecticide is applied to lawns than before.

Once a decision has been made to apply a pesticide and an appropriate control is selected, then it is necessary to determine whether to broadcast or spot treat the infested lawn. Pest populations are usually clumped in certain areas and are not uniformly distributed throughout the lawn. Insects such as caterpillars or mole crickets may occur together soon after hatching, but as food availability decreases and competition increases, individuals may spread out. The amount of insecticide used can be greatly reduced if only the infested locations and perhaps a small buffer area are treated. Along with the reduction in the amount of insecticide, spot treatment reduces the exposure of non-target organisms to the insecticide.

CONCLUSIONS

Overuse of insecticides in urban areas is risky for several reasons, and can be avoided if several other non-chemical controls have been implemented.^[15] People, pets, wildlife, beneficials, and other non-target organisms may be exposed to insecticides that have not been properly applied. Such exposure could cause acute or chronic health problems. Insecticides could leach through the soil or enter groundwater through gutters or drainage areas and result in non-point source pollution. Frequent insecticide use against particular pests may result in resistant pest populations that are even harder to control. And, because some insecticides kill beneficial organisms, their use may increase the risk of pest resurgences or secondary pest outbreaks. However, responsible insecticide use, in combination with other Integrated Pest Management (IPM) tactics, results in healthier lawns with few or no negative environmental consequences.

REFERENCES

1. Beard, J.B.; Green, R.L. The role of turfgrasses in environmental protection and their benefits to humans. *J. Environ. Quality* **1994**, *23* (2), 452–460.
2. Potter, D.A. *Destructive Turfgrass Insects: Biology, Diagnosis, and Control*; Ann Arbor Press: Chelsea, MI, 1998.

3. Tashiro, H. *Turfgrass Insects of the United States and Canada*; Cornell University Press: Ithaca, New York, 1987.
4. Crutchfield, B.A.; Potter, D.A. Tolerance of cool-season turfgrasses to feeding by Japanese beetle and southern masked chafer (Coleoptera: Scarabaeidae) grubs. *J. Econ. Entomol.* **1995**, *88* (5), 1380–1387.
5. Brandhorst-Hubbard, J.L.; Flanders, K.L.; Appel, A.G. Oviposition site and food preference of the green June beetle (Coleoptera: Scarabaeidae). *J. Econ. Entomol.* **2001**, *94* (3), 628–633.
6. Lynch, R.E.; Martin, P.B.; Garner, J.W. Cultural manipulation of coastal bermudagrass to avoid losses from the fall armyworm. *Fla. Entomol.* **1980**, *63* (4), 411–419.
7. Chang, N.T.; Wiseman, B.R.; Lynch, R.E.; Habeck, D.H. Influence of N fertilizer on the resistance of selected grasses to fall armyworm larvae. *J. Agric. Entomol.* **1985**, *2* (2), 137–146.
8. Dahms, R.G. The effect of fertilizers on chinch bug resistance in sorghums. *J. Econ. Entomol.* **1940**, *33* (4), 688–692.
9. Potter, D.A.; Powell, A.J.; Spicer, P.G.; Williams, D.W. Cultural practices affect root-feeding white grubs (Coleoptera: Scarabaeidae) in turfgrass. *J. Econ. Entomol.* **1996**, *89* (1), 156–164.
10. Hertl, P.T.; Brandenburg, R.L.; Barbercheck, M.E. Effect of soil moisture on ovipositional behavior in the southern mole cricket (Orthoptera: Gryllotalpidae). *Environ. Entomol.* **2001**, *30* (3), 466–473.
11. Williamson, R.C.; Potter, D.A. Oviposition of black cutworm (Lepidoptera: Noctuidae) on creeping bentgrass putting greens and removal of eggs by mowing. *J. Econ. Entomol.* **1997**, *90* (2), 590–594.
12. Davis, M.G.K.; Smitley, D.R. Association of thatch with populations of hairy chinch bug (Hemiptera: Lygaeidae) in turf. *J. Econ. Entomol.* **1990**, *83* (6), 2370–2374.
13. Majeau, G.; Brodeur, J.; Carrière, Y. Lawn parameters influencing abundance and distribution of the hairy chinch bug (Hemiptera: Lygaeidae). *J. Econ. Entomol.* **2000**, *93* (2), 368–373.
14. Potter, D.A. Effects of pesticides on beneficial invertebrates in turf. In *Handbook of Integrated Pest Management for Turf and Ornamentals*; Leslie, A.R., Ed.; Lewis Publishers: New York, 1994; 59–70.
15. Leslie, A.R. *Handbook of Integrated Pest Management for Turf and Ornamentals*; Lewis Publishers: New York, 1994.

Insecticide Resistance Management

Edward J. Grafius

Department of Entomology, Michigan State University, East Lansing, Michigan, U.S.A.

INTRODUCTION

Insecticide resistance is one of the most severe problems facing managers of agricultural systems. Insecticide resistance can be defined as an increase of greater than 10-fold in tolerance to an insecticide, or adaptation by the insect to an insecticide resulting in the loss of effectiveness of the insecticide in the field. Resistance may result in increased control costs and crop loss, reduced effectiveness of insecticides, and loss of previously effective products. Even if new insecticides are developed to replace products lost due to resistance, control costs are almost certainly going to be higher than previous ones.^[1]

Insecticide resistance is the direct result of an evolutionary process of selection initiated by the application of insecticide. Insects develop resistance in proportion to the intensity of selection pressure and the pests' genetic resources. In agricultural systems, selection pressure is often very intense because of the need, or perceived need, for very low pest numbers and the lack of alternatives to insecticides for control. Pests often have a wide background of genetic resources available to resist pesticides because of their long evolutionary history of adapting to plant allelochemicals. In fact, some plant chemicals, such as alkaloids found in potatoes, are cholinesterase inhibitors just like many of our insecticides.

One of the first reports of resistance is the San José scale resistance to sulfur lime.^[2] Melander begins our long discussion about managing resistance, noting the genetic nature of the observed resistance, possible genetic variability within the pest population, the importance of refugia, and the potential use of insecticide alternations and mixtures.

INSECTICIDE USE TO MINIMIZE RESISTANCE

Strategies to manage insecticide resistance nearly always emphasize tactics to optimize the use of insecticides. These tactics are readily adoptable and fit within the normal production practices. Tactics include insecticide mixtures, alternation of insecticides, and use of high doses. Each of these tactics has a basis in theory; however, they rely on assumptions often not met for the more problematic pests.^[3,4]

The use of insecticide mixtures assumes that it will be difficult for a single insect to have genes contributing resistance to both insecticides; either the fitness cost of resistance to each insecticide is high and the cost of carrying resistance to two insecticides would be very high, or resistance between the two insecticides is negatively correlated and an insect cannot be resistant to both (e.g., if resistance to the two insecticides were two different forms of the same detoxification enzyme, or involved alterations of the same target site). Mixtures have been proven to be useful in some situations, but unfortunately, it is very common for insects to carry resistance to multiple insecticides with little or no fitness cost to resistance. Thus, although useful in some situations, the use of mixtures carries a significant risk that the target insect will rapidly become resistant to both insecticides.

Alternation of insecticides as a resistance management tactic assumes that resistance to an insecticide will decrease in the absence of selection. In theory, if insecticide A is used for one or several generations of the pest, resistance to B will decrease. This assumes that there is a fitness cost to resistance or some other reason for instability, or there is negatively correlated cross-resistance between insecticides. Alternations must be applied to different insect generations, not within the same generation—or else the practice becomes another form of mixture. In addition, alternations must be between insecticides that have different modes of action or, even better if this is known, between insecticides with different potential mechanisms of resistance. For example, alternating between two insecticides that act as cholinesterase inhibitors is likely to be ineffective, especially if the mode of resistance is insensitive acetylcholinesterase or a general microsomal oxidase detoxification system.

Again, the assumptions underlying the use of alternations are often false; resistance to insecticides is often extremely stable, or at least decreases slowly in the absence of selection, so that one or a few generations without selection do not result in a significant return toward susceptibility. An insect's ability to maintain resistance to multiple insecticides also acts against this tactic for resistance management. However, in some cases, it appears that alternation of insecticides may increase the length of effective control. Thus, for alternation of insecticides, the possible adverse effects do

not seem to be as serious as with mixtures, and there may be situations where it is helpful. Alternation of insecticides is a common recommendation for the management of insecticide resistance and may help in some situations. However, at best, alternation will slightly increase the time to control failure, not greatly lengthen the time or avoid problems entirely.

Use of high doses of insecticides in conjunction with structured refugia has a strong basis in theory and may also hold promise for managing insect pest adaptation to genetically engineered resistant crop varieties.^[5-9] The high-dose tactic assumes that a high dose of insecticide will kill both susceptible individuals and heterozygous individuals (resistance is recessive, or at least partially recessive, so that a high-enough dose will kill heterozygotes). A second key assumption is that initial resistance gene frequency is low; therefore there are extremely few homozygous resistant individuals.^[6] The high-dose tactic also assumes that the high dose of the insecticide can be uniformly distributed throughout the crop and that a high dose does not decay to a low dose, which would selectively kill homozygous susceptible individuals but not heterozygous individuals; these assumptions are difficult or impossible to meet with traditional insecticides, but can perhaps be met at least partially in the case of genetically engineered resistant crops.

IMPORTANCE OF STRUCTURED REFUGIA

An essential component of the high-dose resistance management tactic is the presence of structured refugia for susceptible individuals.^[6,8,9] This refugia provides a source of susceptible individuals so that the gene frequency for resistance will continue to remain very low. Essential aspects of refugia for resistance management are: the refuge must produce large numbers of insects, and there must be a high degree of gene flow between the refuge and the crop, with gene flow occurring before mating. Refugia would be ineffective if resistance was dominant and heterozygous individuals were resistant. A combination of high dose and structured refugia is the basis for many programs to manage pest adaptation to genetically engineered crops containing *Bacillus thuringiensis* toxins.^[10]

INSECTICIDE RESISTANCE IN COLORADO POTATO BEETLE—AN EXTREME CASE

For difficult-to-manage insects such as the Colorado potato beetle, many of the resistance management assumptions for the use of mixtures, alternations, or high doses plus structured refugia are invalid. For example, the Colorado potato beetle is notorious for

its ability to express resistance to multiple insecticides from diverse insecticide groups.^[11] Insecticide resistance in the Colorado potato beetle is known to involve a wide variety of different mechanisms, including microsomal oxidase-based and esterase-based detoxification, insensitive acetylcholinesterase, reduced penetration, and sequestration;^[11] there often appears to be little or no fitness cost to this resistance, and individual insects may express multiple forms of resistance. In addition, resistance to many insecticides in the Colorado potato beetle is stable for long periods. Resistance in the Colorado potato beetle is often dominant or partially dominant, and often imparts such high levels of resistance even in heterozygotes that a high-dose/refugia strategy is impossible.^[12]

RESISTANCE MONITORING AND THE USE OF SIMULATION MODELS

Resistance monitoring is an essential tool of resistance management. Unfortunately, monitoring is often so costly and time-consuming that with even the simplest of systems,^[13] large numbers of individuals cannot be monitored and resistance cannot be detected until resistance gene frequencies are relatively high (e.g., >1%) and may be beyond our ability to manage. On-farm observations of control efficacy as a part of crop monitoring and integrated pest management (IPM) may be the best way to monitor for the initial appearance of resistance. This may be especially true for genetically engineered resistant crop varieties; the host plant resistance factor is uniformly expressed in the crop and any surviving insects are almost certainly resistant.

The development of sophisticated computer models of resistance offers a powerful tool for us to test various management tactics and strategies. These models can include multiple resistance genes, refugia, different selection pressures, different gene flow rates, etc. However, model results are highly dependent on initial inputs of initial resistance gene frequency, intensity of selection, dominance of resistance genes, gene flow between crop and refugia, etc. These factors are often difficult to measure and information may be available only after resistance occurs—too late to help design proactive resistance management strategies.^[14]

PRACTICAL CONSIDERATIONS FOR RESISTANCE MANAGEMENT

The practical limitations of resistance management are also important. For example, the use of a refugia assumes a large number of insects produced in the refuge and migrating into the target crop. This may pose unacceptable pest damage and refuge/crop losses,

for example, in horticultural crops where virtually no contamination by insects is allowed, or for highly valuable nursery plantings. Another common occurrence for problematic pests is that insecticides are only available one at a time, as fast as they become registered and then lose effectiveness, and alternations or mixtures may not be possible.

Another practical factor of resistance management is that it must be initiated before problems appear and must be practiced on a regional basis, not on a farm-by-farm basis.^[15] The action of one or a few individual managers can create a resistance problem despite sound pest management practices by the majority of farmers in a region. Regional cooperation is much more likely in industries made up of a few large farmers than in industries comprising many small, independent farmers.^[16] Those most likely to join a resistance management program are growers who have already experienced serious crop losses due to resistance. However, even in these cases, there can be serious misunderstandings about resistance and resistance management. The farm manager may believe that if one can kill all pest individuals, then none will survive to reproduce and create a resistance problem. A common reaction to resistance is higher and higher insecticide application rates, more and more frequent applications, and intensive use of insecticide mixtures—all measures that can contribute to the most rapid selection for resistance.

IMPORTANCE OF INTEGRATED PEST MANAGEMENT IN RESISTANCE MANAGEMENT

Reducing selection pressure is the best and surest way to manage insecticide resistance. Effective resistance management in the field almost always involves a reduced frequency of application of the insecticide(s).^[17,18] Reduced insecticide application frequencies and reduced selection pressure are most often achieved through IPM, including resistant varieties, biological control, crop rotation, crop scouting and economic thresholds, mass trapping, pheromone disruption of mating, etc.^[19] It is perhaps impossible to manage insecticide resistance in problematic insects such as the Colorado potato beetle, diamondback moth, cotton bollworm, aphid species, whitefly species, etc.—which solely rely on chemical management tactics—without the introduction of IPM technologies.

The U.S. Environmental Protection Agency now recommends that resistance management considerations be included in official pesticide labels,^[20] including the chemical management tactics discussed above as well as IPM-based management and monitoring for resistance. A widespread adoption of IPM strategies

will decrease selection pressure for resistance to insecticides. It is absolutely essential that we conserve current and future insecticides because this resource is limited and new products are costly to develop.

CONCLUSION

The introduction of genetically engineered resistant crop varieties could be an important step toward reducing the impact of insecticide resistance, but in turn, these technologies also need to be protected from the proven adaptability of pests. A regulatory approach to resistance management may be necessary, but even intensive regulation, as occurs in the European Union, does not ensure compliance.^[21] Voluntary adoption of integrated, multiple, tactic management strategies, with the multiple benefits associated with these strategies, will allow the continued use and effectiveness of new and traditional insecticides and of genetically engineered resistant crop varieties.

REFERENCES

1. Grafius, E. Economic impact of insecticide resistance in Colorado potato beetle (Coleoptera: Chrysomelidae) on the Michigan potato industry. *J. Econ. Entomol.* **1997**, *90*, 1144–1151.
2. Melander, A. Can insects become resistant to sprays? *J. Econ. Entomol.* **1914**, *7*, 167–173.
3. Denholm, I.; Rowland, M. Tactics for managing pesticide resistance in arthropods: theory and practice. *Annu. Rev. Entomol.* **1992**, *37*, 91–112.
4. Leeper, J.; Roush, R.; Reynolds, H. Preventing or managing resistance in arthropods. In *Pesticide Resistance: Strategies and Tactics for Management*; National Academy Press: Washington, DC, 1986; 335–346.
5. Tabashnik, B.; Croft, B. Managing pesticide resistance in crop–arthropod complexes: interactions between biological and operational factors. *Environ. Entomol.* **1982**, *11*, 1137–1144.
6. Roush, R. Managing pests and their resistance to *Bacillus thuringiensis*: can transgenic crops be better than sprays? *Biocontrol Sci. Technol.* **1994**, *4*, 501–516.
7. Roush, R. Can we slow adaptation by pests to insect transgenic crops?. In *Biotechnology and Integrated Pest Management*; Persley, G., Ed.; CABI: London, 1996; 242–263.
8. Gould, F. Sustainability of transgenic insecticidal cultivars: integrating pest genetics and ecology. *Annu. Rev. Entomol.* **1998**, *43*, 701–726.
9. Shelton, A.; Zhao, J.; Roush, R. Economic, ecological, food safety, and social consequences of the deployment of Bt transgenic plants. *Annu. Rev. Entomol.* **2002**, *47*, 845–881.
10. U.S. EPA; Office of Pesticide Programs. *Revised Bt Crops Assessment*; 2001. www.epa.gov/pesticides/

- biopesticides/otherdocs/bt7lowbar;reassess/5-Insect%20Resistance%20Management.pdf.
11. Bishop, B.; Grafius, E. Insecticide Resistance in the Colorado Potato Beetle. In *Chrysomelidae Biology*; 1996; 1, 355–377.
 12. Ioannidis, P.; Grafius, E.; Wierenga, J.; Whalon, M.; Hollingworth, R. Selection, inheritance and characterization of carbofuran resistance in the Colorado potato beetle (Coleoptera: Chrysomelidae). *Pestic. Sci.* **1992**, *35*, 215–222.
 13. Bishop, B.; Grafius, E. An on-farm insecticide resistance test kit for Colorado potato beetle (Coleoptera: Chrysomelidae). *Am. Potato J.* **1991**, *68*, 53–64.
 14. Roush, R.; McKenzie, J. Ecological genetics of insecticide and acaricide resistance. *Annu. Rev. Entomol.* **1987**, *32*, 361–380.
 15. Pimentel, D.; Burgess, M. Effects of single versus combinations of insecticides on the development of resistance. *Environ. Entomol.* **1985**, *14*, 582–589.
 16. Jutsum, A.; Heaney, S.; Perrin, B.; Wege, P. Pesticide resistance: assessment of risk and the development and implementation of effective management strategies. *Pestic. Sci.* **1998**, *54*, 435–446.
 17. Dennehy, T.; Williams, L. Management of resistance in *Bemisia* in Arizona cotton. *Pestic. Sci.* **1997**, *51*, 398–406.
 18. Horowitz, A.; Mendelson, Z.; Cahill, M.; Denholm, I.; Ishaaya, I. Managing resistance to the insect growth regulator, pyriproxyfen, in *Bemisia tabaci*. *Pestic. Sci.* **1999**, *55*, 272–276.
 19. Hull, L.; McPheron, B.; Lake, A. Insecticide resistance management and integrated mite management in orchards: Can they coexist?. *Pestic. Sci.* **1997**, *51*, 359–366.
 20. U.S. EPA; Office of Pesticide Programs. *Pesticide Registration Notice PR 2001-5*; 2001. www.epa.gov/oppmsd1/PR_Notices/pr2001-5.pdf.
 21. Clarke, J.; Clark, W.; Hancock, M. Strategies for the prevention of development of pesticide resistance in the UK—lessons for and from the use of herbicides, fungicides and insecticides. *Pestic. Sci.* **1997**, *51*, 391–397.

Integrated Pest Management: Principles with Emphasis on Weeds

Heinz Müller-Schaerer

Département de Biologie/Ecologie, Université de Fribourg/Perolles,
Fribourg, Switzerland

INTRODUCTION

Agrochemical companies promise that transgenic crops will simplify pest management programs through the use of singular chemical tactics. This “silver-bullet” approach has consistently failed and almost certainly will again. It will do so as a result of fundamental ecological relationships governing population size and diversity.^[1] At the same time, in many countries, pesticide policies have called for significant use reductions together with the promotion of biodiversity in agro-ecosystems.^[2] However, initiatives to reduce reliance on herbicides will require a much fuller understanding of how management practices complement one another to maintain weed populations at low equilibrium densities. Biological control approaches require, but also provide, detailed insight into weed-crop interactions and how they are influenced by both the biotic and abiotic environments. They can, thus, be viewed as the basis for integrated production.^[3] In most cases, only combinations with other weed management tools will result in acceptable levels of weed control. Various types of integration can be envisaged, of which preventative measures will be most important for developing sustainable agricultural production.

WEED CONTROL, WEED SCIENCE, AND INTEGRATED WEED MANAGEMENT

At the close of the twentieth century, agricultural weed management is diverging in two distinct directions. In one set of farming systems, farmers rely primarily on herbicides to suppress weeds. This approach is exemplified by the extensive maize (*Zea mays* L.)/soybean (*Glycine max* (L.) Merr.) system of the midwestern United States, where >110 million kg of herbicide active ingredients are applied annually to >95% of the area planted with those two crops. In a second set of farming systems, herbicides are largely or entirely avoided, and weeds are mainly suppressed using physical and ecological tactics. The existence, and risk of development, of herbicide resistance makes herbicide-dependent cropping systems increasingly vulnerable.

Moreover, widespread concern about environmental side effects of herbicides combined with fear for public health, has resulted in several herbicides being banned in some countries and increasing pressure on farmers to reduce the use of herbicides.^[4]

In contrast to disciplines of plant pathology and entomology, the “how to control” technological orientation was shaped early on in the evolution of weed science as a discipline and, until recently, this has dominated the science. The fact that weeds have been regarded as a problem that can be controlled with herbicides, rather than managed through cropping system design, has resulted in a time lag in developing integrated weed management systems, as compared to integrated pest and disease management systems.^[1] The United Nations Conference on Environment and Development (UNCED), in its Agenda 21, recognized integrated pest management (IPM) as the preferred strategy to achieve sustainable agricultural production. IPM typically involves a reduction in the reliance on chemical pesticides, including herbicides. Furthermore, in the Convention on Biological Diversity, the point is clearly made that priority should be given to biological control as a component of future pest management.

METHODS USED TO CONTROL CROP WEEDS BIOLOGICALLY

Three principal methods of biological weed control can be distinguished (Fig. 1):^[3,5] 1) The “inoculative” or “classical” approach aims to control naturalized weeds by the introduction of exotic control organisms from the weed’s native range. They are released over only a small area of the total weed infestation and control is achieved gradually. Successful control depends on favorable conditions promoting an increase in the control agent’s population, establishment of epiphytotics and, so, reduction of the target weed population. 2) The “inundative” or “bioherbicide” method uses periodic releases of an abundant supply of the control agent over the entire weed population to be controlled. Such biological agents generally are manufactured, formulated, standardized, packaged, and registered

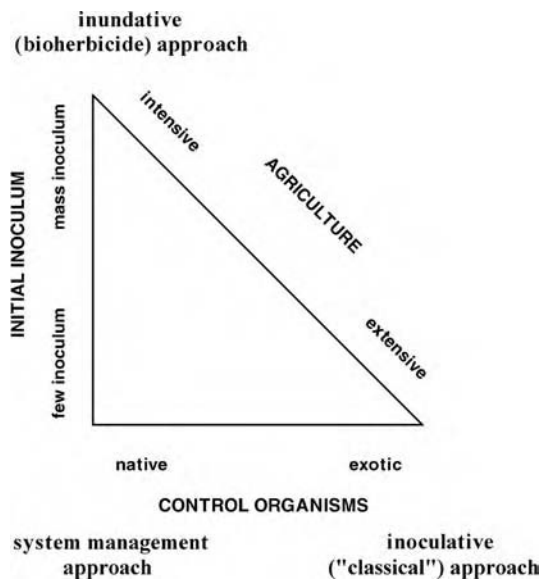


Fig. 1 Methods of biological weed control in agro-ecosystems.

like chemical herbicides. Compared to the other two approaches, this approach is characterized by higher application costs and a relatively short time period to achieve a potential control success. 3) More recently, the “system management approach” of biological weed control had been described. It is related to the conservation and augmentative approaches distinguished by some authors. Its aim is to shift the competitive weed–crop relationship in favor of the latter, mainly by stimulating the buildup of a disease epidemic or insect outbreak on the target weed population. The approach excludes the use of exotic organisms (classical approach) and the use of mass amounts of inoculum applied like a herbicide to the whole weed population (bioherbicide approach).

INTEGRATING BIOLOGICAL CONTROL WITH OTHER METHODS OF WEED MANAGEMENT

Weed problems in agro-ecosystems are rarely caused by single weed species. Clearly, biological control, with its inherently narrow spectrum, has to be considered as an integrated component of a well-designed pest management strategy, not as a cure by itself. In most cases, combinations of biological agents with other weed management tools will be needed to produce acceptable levels of overall weed control. Such integration can be viewed as a vertical integration of various control tactics against a single weed species, or as a horizontal integration across different weed species in one crop^[6] (Table 1). Horizontal integration mainly involves the combination of microbial herbicides with

chemical herbicides or mechanical methods to broaden the spectrum of weed species controlled. Furthermore, in situations where particularly high doses of herbicides are needed to control a single weed species while the rest of the weed flora could be controlled by lower amounts, biological control may allow considerable reduction of herbicide inputs and contribute to maintaining species diversity in crops. Three possible types of vertical integration of biological control with other methods of weed management can be distinguished, both in time and space: purpose-specific approaches, ecological integration, and physiological integration^[7] (Table 1).

Purpose-Specific Approaches

The type and level of control are chosen according to the requirements. This often involves different methods to be applied at different sites. For instance, for a weed that is still spreading, chemical herbicides may well be the method of choice to remove new infestations, while biological control may be relied on to give long-term control of large, established infestations.^[11]

Ecological Integration

This term is given to situations where different approaches are used often at the same time on the same infestation. Integration with herbicides^[12,13] and with plant (crop) competition^[10,14,15] is most widely envisaged. This type of integration essentially summarizes holistic approaches that encompass all modifications to the environment, which may favor the effectiveness of biological control agents and facilitate the management of a weed population.^[16]

Physiological Integration

This type of integration exploits synergistic interactions between changes in the biochemistry of weeds, often produced by sublethal effects of herbicides and the effectiveness of biological control agents. Herbicides (or other “synergists”) are known to increase incidence of infection and to enhance the growth of pathogens,^[17–19] but infection by the pathogen may also facilitate the uptake of herbicides, mainly by injuring the cuticle and epidermis of the host. In addition, various studies have shown greatly increased disease severity and agent effects when combined with phytotoxic metabolites produced by the pathogen,^[20] or with specific formulation and delivery techniques of microbial herbicides.^[21] Thus, physiological integration is directed toward combined effects with biological control agents on plant individuals.

Table 1 Selected examples of successful integrated weed management involving biological control

| Target weed | Crop/habitat | Control methods | Type of integration ^a |
|--|-------------------------|---|--|
| Northern jointvetch (<i>Aeschynomene virginica</i>) | Rice and soybean fields | Pathogen–pathogen–herbicide | Horizontal (PSA ^b) |
| Stanglervine (<i>Morrenia odorata</i>) | Citrus groves | Pathogen–herbicide | Horizontal |
| Velvetleaf (<i>Abutilon theophrasti</i>) | Corn, soybean | Pathogen–herbicide | Horizontal; vertical/physiological (PSA) |
| Barnyard grass (<i>Echinochloa crus-galli</i>) | Various crops | Pathogen–herbicide | Vertical/physiological |
| Nutsedges (<i>Cyperus</i> spp.) | Various crops | Pathogen–herbicide | Vertical/physiological (PSA) |
| Spotted knapweed (<i>Centaurea maculsa</i>) | Rangeland | Insect–plant competition Insect–herbicide | Vertical/ecological Vertical/physiological, or purpose-specific |
| Nodding thistle (<i>Carduus nutans</i>) | Rangeland | Insect–herbicide Insect–plant competition | Vertical/physiological Vertical/ecological |
| St. John’s wort (<i>Hypericum perforatum</i>) | Rangeland | Insect–fire Insect–plant competition | Vertical/ecological Vertical/ecological |
| Water hyacinth (<i>Eichhornia crassipes</i>) | Aquatic | Insect–herbicide Insect–pathogen–herbicide | Vertical/physiological, or purpose-specific |
| Floating fern (<i>Salvinia molesta</i>) | Aquatic | Insect–herbicide Insect–fertilizer | Vertical/purpose-specific, or physiological Vertical/physiological |

^aSee text for details.

^bPSA: partially sequential application.
(From Refs.^[6–10].)

Ultimately, optimal management, with minimal disruptive interventions, requires a good understanding of the weed’s biology and, especially, population dynamics.^[22] Biological weed control requires, and provides, a detailed ex-ante analysis of the problem situation, especially of the crop environment, revealing interactions between the various components and their underlying interactions. It should, therefore, be the strategy that is basic to integrated production systems. Bridges between different disciplines need to be built to optimize the fit of biological control into existing management systems.^[3,7]

FUTURE DIRECTIONS

When weeds are no longer regarded as a problem to be resolved by curative tactics, then prevention becomes the keyword and integrated cropping management the new concept, of which integrated weed management is an important component. To integrate soil, crop, and weed management effectively, much work remains to be done by scientists spanning a broad range of disciplines.^[4] In parallel, to transfer the scientific knowledge into farming practices, a considerable amount of time

must be spent with farmers in order to understand the true practical dimensions of the increasingly complex study systems. In this cropping system design approach, numerous fitness-reducing and mortality events are integrated to manage weed populations, with herbicides being used as a last resort. Prevention involves any aspect of management that favors the crop relative to the weed. This includes the development of competitive crop cultivars, crop rotation, mixed cropping, and allelopathy.^[23] Preventative control requires a detailed insight into weed biology and ecology and the ways in which they interact with the crop. Biological control provides a fundamental tool for successful management of weed populations, where weed control no longer aims at crop production in a weed-free environment, but simply at a reduction of weed-induced yield losses. By that, it greatly contributes to promoting biodiversity in human-influenced landscapes, a central pillar of modern, sustainable agriculture.

REFERENCES

1. Mortensen, D.A.; Bastiaans, L.; Sattin, M. The role of ecology in the development of weed management systems: an outlook. *Weed Res.* **2000**, *40* (1), 49–62.

2. Scheepens, P.C.; Müller-Schärer, H.; Kempenaar, C. Opportunities for biological weed control in Europe. *BioControl* **2001**, *46* (2), 127–138.
3. Müller-Schärer, H.; Scheepens, P.C.; Greaves, M.P. Biological control of weeds in European crops: recent achievements and future work. *Weed Res.* **2000**, *40* (1), 83–98.
4. Liebman, M.; Davis, A.S. Integration of soil, crop and weed management in low-external-input farming systems. *Weed Res.* **2000**, *40* (1), 27–47.
5. Müller-Schärer, H.; Frantzen, J. An emerging system management approach for biological weed control in crops: *Senecio vulgaris* as a research model. *Weed Res.* **1996**, *36* (6), 483–491.
6. Watson, A.K.; Wymore, L.A. Biological control, a component of integrated weed management. In *VII International Symposium of Biological Control of Weeds*; Delfosse, E.S., Ed.; Ist. Sper. Patol. Veg. (MAF): Rome, Italy, 1989; 101–106.
7. Cullen, J.M. Integrated control and management. In *Proceedings of the IX International Symposium on Biological Control of Weeds*; Moran, V.C., Hoffman, J.H., Eds.; University of Cape Town, South Africa: Stellenbosch, South Africa, 1996; 483–486.
8. Charudattan, R. Biological control of weeds by means of plant pathogens: significance for integrated weed management in modern agro-ecology. *BioControl* **2001**, *46* (2), 229–260.
9. Julien, M.H.; Griffiths, M.W. Biological control of weeds. In *World Catalogue of Agents and Their Target Weeds*, 4th Ed.; CABI Publishing: Wallingford, UK, 1998.
10. Sheppard, A.W. The interaction between natural enemies and interspecific plant competition in the control of invasive pasture weeds. In *IX Int. Symposium on Biological Control of Weeds*; Moran, V.C., Hoffman, J.H., Eds.; Stellenbosch, South Africa, 1996; 19–26.
11. Müller-Schärer, H.; Schroeder, D. The biological control of *Centaurea* spp. in North America: do insects solve the problem? *Pestic. Sci.* **1993**, *37*, 343–353.
12. Scheepens, P.C. Joint action of *Cochliobolus lunatus* and atrazine on *Echinochloa crus-galli* (L.). *Weed Sci.* **1987**, *27*, 43–47.
13. Wymore, L.A.; Watson, A.K.; Gotlieb, A.R. Interaction between *Colletotrichum coccodes* and thidiazuron for control of velvetleaf (*Abutilon theophrasti*). *Weed Sci.* **1987**, *35*, 377–383.
14. DiTommaso, A.; Watson, A.K.; Hallett, S.G. Infection by the fungal pathogen *Colletotrichum coccodes* affects velvetleaf (*Abutilon theophrasti*)–soybean competition in the field. *Weed Sci.* **1996**, *44*, 924–933.
15. Müller-Schärer, H.; Rieger, S. Epidemic spread of the rust fungus *Puccinia lagenophorae* and its impact on the competitive ability of *Senecio vulgaris* in celeriac during early development. *Biocontrol Sci. Technol.* **1998**, *8* (1), 59–72.
16. Newman, R.M.; Thompson, D.C.; Richman, D.B. Conservation strategies for the biological control of weeds. In *Conservation Biological Control*; Barbosa, P., Ed.; Academic Press: San Diego, USA, 1998; 371–396.
17. Hasan, S.; Ayres, P.G. The control of weeds through fungi: principles and prospects. *New Phytol.* **1990**, *115*, 201–222.
18. Sharon, A.; Amsellem, Z.; Gressel, J. Glyphosate suppression of an elicited defence response. Increased susceptibility of *Cassia obtusifolia* to a mycoherbicide. *Plant Physiol.* **1992**, *98*, 54–659.
19. Gressel, J., et al. Biocontrol of weeds: overcoming evolution for efficacy. *J. Environ. Sci. Health, Part B* **1996**, *31* (3), 399–404.
20. Vurro, M.; Bottalico, A.; Capasso, R.; Evidente, A. Cytochalasins from Phytopathogenic Ascochyta and Phoma Species. In *Toxins in Plant Disease Development and Evolving Biotechnology*; Mukherji, K.G., Upadhyay, R.K., Eds.; IBH Publishing Co. Pvt, Ltd.: Oxford, 1997.
21. Greaves, M.P. Microbial herbicides—factors in development. In *Crop Protection Agents from Nature: Natural Products and Analogues*; Copping, L.G., Ed.; Royal Society of Chemistry: Cambridge, UK, 1996; 444–467.
22. Cousens, R.; Mortimer, M. *Dynamics of Weed Populations*; Cambridge University Press: London, 1995; 332 pp.
23. Zimdahl, R.L. *Fundamentals of Weed Science*; Academic Press, Inc.: San Diego, USA, 1993; 450 pp.

Integrated Plant Control: System and Management

Ján Gallo

Department of Plant Protection, Faculty of Agronomy, Slovak Agricultural University, Nitra, Slovak Republic

INTRODUCTION

Measures taken for the protection of plants against harmful organisms (diseases, animal pest, and weeds) can influence significantly other components of the living environment. Therefore, it is of utmost importance to elaborate and to employ a system that not only protects plants effectively, but also takes into account economical and ecological factors. The endeavor to develop such a system has resulted in the concept of integrated pest management (IPM). This article provides an overview of IPM in Central Europe and represents both whole and open systems.

DEFINITION AND HISTORY

The definition—but mainly the content—of “IPM” is a favorite topic of discussions not only among experts, but in the popular media as well. This notion is used for free and evokes a semblance of progress without appropriately understanding the essence of the matter (Refs.^[1,2] see also Ref.^[3]).

More than 20 years ago, the chemical fight against all harmful organisms dominated crop protection, and chemicals were reliable and, to some extent, economically advantageous as well. However, plant protection based merely on chemical methods caused some problems, including pesticide residues, formation of resistance of pests against pesticides, and hygienic difficulties. Recent knowledge of the various fields of biological research, biochemistry, and agriculture has led to the conclusion that effective long-term maintenance of soil fertility and biocenosis requires ecologically, toxicologically, and economically respectable crop protection measures.

Present-day conceptual intentions of plant protection in all agriculturally developed countries are aimed to ensure plant production and to secure the whole society’s interests in the ecological agriculture as well. The work of a crop protectionist and grower aims to represent ecological integrity.

Agricultural production and plant production operate in nature. For this, each realized agrotechnical measure shows an appropriate or inappropriate effect on respective items of the living environment including

the soil, water, air, and biosphere. Plant control, with its preventive!—but especially repressive—interventions against harmful agents, significantly influences components of the living environment.

Taking into account scientific knowledge and ecological plant control, this resulted in a new concept of plant protection. The system connected economical and rational control based on ecological requirements. Elaboration and submission of integrated pest management resulted in the term IPM, which has been changing with time. Investigating the history of integrated systems, Samersov^[4] concluded that integrated plant control originated in the 1940s of the 20th century in several places at the same time, namely in Canada and California, under the participation of Dr. Pickett and other entomologists who believed in the principle that an ecological view was needed in the solution of pest control. The term “integrated control” and the formation of its principles were submitted by an Italian entomologist, F. Silvestri, in 1932. In the beginning, the goal was to integrate chemical and biological methods in pest control. The authors developed a rational approach to pest control based not only on pesticides, but also on pests’ natural enemies. This conception of pest control appeared in 1959.^[5] Later on, IPM was broadened to include disease and weed control as well.^[2] In the initial period, most of the investigators understood integrated control as an integration of various controlling methods.

The definitions of IPM used by international organizations Food and Agriculture Organization (FAO) and International Organisation for Biological and Integrated Control of Noxious Animals and Plants (IOBC-OILB) dominated the world.

FAO states the definition: “Integrated control represents the system of pest regulation which takes into account respective environment and population dynamics of harmful species and utilizes all suitable techniques and methods in the most effective combination to maintain pest population under the threshold of harmfulness.”

Somewhat different is the definition of IOBC-OILB, which says that, “Integrated control represents procedure (method) which utilizes all economically, ecologically, and toxicologically acceptable methods for keeping the pests under the threshold of harmfulness

with preferential and meaningful utilization of natural restricting factors.’’^[6]

Based on stated works on the topic (see also Ref.^[3]), it is very difficult to imagine what integrated control is and, especially, what its prospects are, how it can positively contribute to agriculture, and where the boundaries of its possibilities are.

Integrated pest management as a scientific discipline must have its own strategy and tactics, scientific goals, and scientifically reasonable definition. Only these can guarantee that IPM will develop, be updated by new scientific discoveries, and be an acquisition for agriculture at the same time.

The essential strategy of IPM is the prognosis of expected losses and the determination of economical thresholds of individual harmful organism species. Integrated pest management strategy is based on the recognition of the effectiveness of natural regulating factors, and is not aimed at the complete eradication of harmful organisms but at the regulation of their populations based on certain ecologically and economically justified levels. Tactics of IPM in agricultural crops represent the utilization of the most rational controlling methods and their combinations directed against harmful species or their complexes. Within this framework, it is emphasized that chemical control plays an important role in integrated systems of plant control.^[4] The aims of IPM are as follows:

- Regulate harmful organism populations and keep them under economical threshold.
- Take into account ecological aspects. All measures have to be applied without breaking ecological balance in the agroecosystem.
- Emphasize the meaning of antagonists (parasitoids, predators, and pathogens) in the control of pests. It is necessary to know the activity and function of antagonists in the system to maximize their effect on pest populations.
- Utilize interdisciplinary, systemic approaches integrating knowledge of various scientific fields.

A practical consequence of these goals is that IPM, as such, does not stand in the foreground of our interest, but protection against damages and losses caused by harmful organisms (diseases, animal pests, and weeds) does.

To realize a system of IPM, we need first of all to study the biological, ecological, toxicological, and economical processes connected with the growing of agricultural crops, as well as the population dynamics of pests and possible controlling tactics that utilize agrotechnical, biological, and chemical methods and a wider introduction of resistant crop varieties.

However, the fact that integrated systems and methods elaborated for respective species of pests or

crops are hardly or only partly utilizable against other harmful pests must be taken into account.^[1]

These concepts of IPM are most precisely expressed by the definition of Hron,^[2] who stated that IPM in agricultural production represents scientifically managed control of cultural plants against negative effects of all types of pests (both biotic and abiotic), which result in the reduction of quality and quantity of crop production. It is an intrinsic part of the agrotechnics of all crops and, for this reason, it must be realized against harmful organisms and agents based on their:

- Diagnosis.
- Prognosis and signalization.
- Complex control (both preventive and repressive).

The respective components of IPM must be systematically interrelated in crop production. These technological components must be continuously updated based on biological, research, and agricultural practices.

DIAGNOSIS

The main role of diagnosis is to recognize, observe, determine, name, and distinguish the pest species. With harmful organisms, the qualitative and accurate determination of the diseases, animal pests, weeds, and defects caused is paramount. Based on sound observations and symptoms, potential damages can be deduced. It provides records and details for symptomatic and etiologial treatment. Appropriate diagnosis is essential in developing control procedures.

The role of the diagnosis of harmful organisms and agents is:

1. To determine harmful organisms in the crop under specific ecological conditions by its accurate identification.
2. To determine the characterization (i.e., reason) and range of harmful effects on plant production, to determine the threshold of harmfulness, and to make a categorization of harmful organisms according to the degree of its harmful effects on crops, stand, and the environment.
3. To elaborate a detailed specification of harmful agents (i.e., *harmful organisms* (biotic)—knowledge about their biological and ethological characteristics; *harmful agents* (abiotic)—determination of physical or chemical principles of harmful effect).
4. To evaluate the situation and make a right decision.
5. To elaborate the record of pest occurrence in crops.

A diagnostic method utilizes various devices, analyses, examinations, mathematical analyses, and procedures to achieve its aims.

PROGNOSIS AND SIGNALIZATION

The effectiveness of crop protection measures, the rational utilization of pesticides, and the protection of the living environment require us to apply a controlling system of agricultural crops based on prognosis and signalization. Prognosis is based on prognostics as a scientific discipline, which represents the most complex and exact information system for the prediction of future events. Thus, in plant protection, prognosis means the prediction of pest occurrence intensity on some stand, locality, or region in advance. Prognosis then predicts some event or the course of events in the future according to the past and present data evaluation. Taking into account as many inside and outside influences and analogies as possible, prognosis searches for the most probable developing trend with causal, statistical, spatial, and time determination of the regions including qualitative and quantitative aspects, and uses methods of detailed biological, technical, chemical, statistical, and mathematical analyses. There are three groups of prognoses distinguished there: several-year prognoses, short-term prognoses, and long-term prognoses.

Signalization

By this notion, we understand operative and topical messages for agricultural enterprises and farms on the necessary protection measures timed optimally and purposefully from the economical, toxicological, and ecological points of view.

COMPLEX CONTROL

Appropriate diagnosis and prognosis, correct signalization, and subsequent evaluation of the situation enable us to realize complex control against harmful organisms. Effective complex control consists of preventive (*indirect, prophylactic*) and repressive (*direct, therapeutic, curative*) protection of cultural field crops, as well as agricultural commodities in stores. Both these components cannot be applied separately because there is a connection and interaction between them.

Preventive Control

The main role of preventive control is protecting healthy plants from pest infection by applying suitable

agrotechnical and special measures. The occurrence of harmful organisms has to be predicted to achieve a control that is profitable and successful.^[1]

Preventive controls include:

1. Correct choice of crop.
2. Appropriate arrangement of crops in crop rotation.
3. Correct tillage of soil.
4. Optimum fertilizer use.
5. Utilization of high-quality seed and correct sowing.
6. Purposeful treatment of plants during vegetation period.
7. Correct harvest of crop and attentive storage of plant products.
8. Special preventive measures including genetic and breeding methods and quarantine measures.^[1,2]

Repressive pest control

Repressive control is usually applied when occurrence, extension, or overdissemination of harmful organisms was not successfully prevented by the methods of preventive control. Direct measures must be specified according to the characteristics of harmful organisms and the extent of its occurrence in crops.^[1]

With repressive control, the following methods are taken into account:

1. Against *harmful agents* (abiotic).
 - a. Biotechnological methods.
2. Against *harmful organisms* (biotic).
 - a. Physical, which involves mechanical and thermal methods.
 - b. Biotechnical and biotechnological, within which a natural reaction of harmful organisms to physical or chemical stimuli are utilized (the so-called biotechnical methods). Biotechnological means are being created by genetic engineering.
 - c. Biological methods, within which humans intentionally utilize organisms (predators, parasites, and parasitoids) to restrict the occurrence of harmful organisms.
 - d. Chemical methods, which utilize pesticides to control the occurrence of harmful organisms.

Based on the abovestated facts, some distinctions can be deduced from up-to-now practical plant control, which was focused mainly on harmful organisms and methods enabling to destroy these organisms or at least reduce their occurrence to irrational minimum. Little attention was devoted to the own host plant and

the relations between two components of the complex host-harmful organism. Usually, several other important components of the ecosystem (as useful or indifferent components of agrocenosis or biocenosis) escape from our attention. Integrated pest management is still being developed and its full realization in farm practice has to be preceded first of all by appropriate theoretical results in research field. (*Nothing is better for practice like a good theory.*)

CONCLUSION

Integrated pest management is inherent in crop production systems. Within the projects “ecological agriculture” and “integrated agricultural production,” the ideas which stress that crop protection against pests cannot be understood separately, but as an integral part of the system, are applied.

The systems of integrated agricultural crop production are considered and discussed. However, one thing must be clear: the systems of integrated agricultural plant production are based on the calculation

of energy balance and include all ecological aspects of IPM, shifting agricultural production from qualified empiricism to scientifically well-founded “terrain” biotechnologies.^[7]

REFERENCES

1. Gallo, J.; Šedivý, J. *Integrovaná Ochrana Rostlín (Integrated Pest Management)*; VES VŠP: Nitra, 1992.
2. Hron, F. Integrovaná ochrana rostlín v zemědělské velkovýrobě ČSSR (IPM in agricultural large-scale production in czechoslovakia). In *Sbor. VŠZ, Fak. Agron., Řada A*, 32; Praha, 1980; 181–198.
3. <http://www.ippc.orst.edu/IPMdefinitions>.
4. Samersov, V.F. *Integrovanaja Sistema Zaščity Zernovykh Kultur ot Vreditel' (IPM of Cereals Against Pests)*; Urodžaj: Minsk, 1988.
5. Stern, V.M.; Smith, R.F.; van den Bosch, R.; Hagen, K.S. The integrated control concept. *Hilgardia* **1959**, *29*, 81–101.
6. Hrdý, I. Insekticídy v integrované ochraně rostlin (Insecticides in IPM). *Agrochémia* **1980**, *20*, 203–214.
7. Hrdý, I. *Biopesticídy v Zemědělství (Biopesticides in Agriculture)*; Praha, MZČR, 1981.

International Pesticide Poisoning Surveillance

Nida Besbelli
Jenny Pronczuk

*International Programme on Chemical Safety, World Health Organization,
Geneva, Switzerland*

INTRODUCTION

In view of the continuing interest expressed in pesticide poisoning by medical and environmental professionals and related sectors, the International Programme on Chemical Safety (IPCS)^a has initiated a range of activities designed to characterize the true extent and severity of pesticide poisoning worldwide and set up the basis for surveillance systems. Based on this interest, on the concern expressed by Member States, and on previous work and recommendations made, the IPCS is promoting formal studies on pesticide exposure and poisoning at country and regional levels. The activities proposed aimed at strengthening the evidence base for health protection by WHO estimate the burden of illness in countries and help prevent and mitigate the effects of pesticides on human health.

OBJECTIVES

The main objective of the project is to prevent poisoning by pesticides and to promote their safe use.^[1] This will be achieved through the following activities:

- Epidemiological study and characterization of toxic exposures.
- Setting up surveillance mechanisms and databases on pesticides.
- Training within the health sector.
- Awareness raising through public education and prevention campaigns.

The specific objectives of the project are to:

- Prepare and maintain a database on pesticide poisoning cases including information on types of pesticides involved, circumstances of poisoning, and the main population groups affected.

- Identify the main pesticides involved in human exposures and/or poisoning incidents and to prepare a list and/or database on pesticides.
- Assess the impact of human pesticide poisoning and exposure in relation to geographical/agricultural characteristics of the areas covered by the study (e.g., crop production, forestry, animal husbandry, etc).
- Publish the results of studies in the form of reports (and scientific papers).
- Issue recommendations for action and set the basis for planning prevention and education activities in cooperation with partners.
- Contribute to the regional and global components of the project.

STUDY DESIGN

Pesticide Exposure Record (PER)

On the basis of the experience gained during pilot data collection studies, a form was prepared in 1997 to record patient data: PER (see Fig. 1). This paper form was reviewed in May 1999 and October 1999, and minor additions were made in accordance with recommendations of potential users in countries in South East Asian (SEA) and Latin American (AMR) regions. Some minor modifications were also made at a New Delhi meeting held in India, January 2001, for the evaluation of the first-stage studies in the SEA Region. For severity grading, the classification system Poisoning Severity Score (PSS)^[2] developed by IPCS in cooperation with the European Commission and the European Association of Poisons Centres and Clinical Toxicologists is used.

Data Sources

Patients exposed to pesticides are usually managed at health care facilities of different levels, ranging from Primary Health Care (PHC) to local, regional, and specialized teaching hospitals. For the first-stage studies

^aThe International Programme on Chemical Safety is a joint collaborative program of the International Labour Organization (ILO), the United Nations Environment Programme (UNEP), and the World Health Organization (WHO).

PESTICIDE EXPOSURE RECORD (Confidential)

| | | | |
|--|--|---|---|
| 1. EXPOSURE TIME AND PLACE | | Record number: / / | |
| Date of consultation: / / | Time elapsed since exp: hs dy ms | City | |
| Date of exposure: / / | Duration of exposure : hs dy ms | Province | |
| 2. COMMUNICATION (Source of information) | | | |
| Name: | Institution: () | Phone: | Data collection date: / / |
| Category of person supplying information: <input type="checkbox"/> Medical <input type="checkbox"/> Paramedical | | Officer's initials: | |
| 3. PATIENT DETAILS | | | |
| Name (Initials): | | Identity N° | |
| Sex: <input type="checkbox"/> M <input type="checkbox"/> F | Age: dy ms yr <input type="checkbox"/> Unknown | If unknown: <input type="checkbox"/> Child <input type="checkbox"/> Adolescent <input type="checkbox"/> Adult | |
| 4. CIRCUMSTANCES OF EXPOSURE (check one, plus "uncertain", if relevant) | | | |
| <input type="checkbox"/> Intentional <input type="checkbox"/> Accidental <input type="checkbox"/> Occupational <input type="checkbox"/> Uncertain <input type="checkbox"/> Unknown () | | | |
| 5. MAIN ACTIVITY AT TIME OF EXPOSURE (check one, or more than one if "Multiple") | | | |
| <input type="checkbox"/> Manufacturing/Formulation | <input type="checkbox"/> By-standing | <input type="checkbox"/> Veterinary Therapy | |
| <input type="checkbox"/> Application in field | <input type="checkbox"/> Transportation | <input type="checkbox"/> Multiple (specify) | |
| <input type="checkbox"/> Public health campaign | <input type="checkbox"/> Mixing/Loading | <input type="checkbox"/> Not relevant | |
| <input type="checkbox"/> Household application | <input type="checkbox"/> Equipment care | <input type="checkbox"/> Other (specify) | |
| <input type="checkbox"/> Field re-entry | <input type="checkbox"/> Human Therapy | <input type="checkbox"/> Unknown | |
| 6. LOCATION OF EXPOSURE (check one) | | | |
| <input type="checkbox"/> Home (urban/periurban) | <input type="checkbox"/> Home (rural) | <input type="checkbox"/> Farm/field | <input type="checkbox"/> Greenhouse <input type="checkbox"/> Unknown |
| <input type="checkbox"/> Garden (urban/periurban) | <input type="checkbox"/> Garden (rural) | <input type="checkbox"/> Public area | <input type="checkbox"/> Storage site <input type="checkbox"/> Other (specify) |
| 7. ROUTE OF EXPOSURE (check main route or more than one, if applicable) | | | |
| <input type="checkbox"/> Oral <input type="checkbox"/> Dermal <input type="checkbox"/> Respiratory <input type="checkbox"/> Ocular <input type="checkbox"/> Unknown <input type="checkbox"/> Other (specify) | | | |
| 8. PRODUCT IDENTITY (add other page(s), if necessary, for each product) | | | |
| Product name(s): | | <input type="checkbox"/> Unknown | (Co-ordinator to fill-in) Use intended: <input type="checkbox"/> Registered <input type="checkbox"/> Not approved |
| Active Ingredient: | | <input type="checkbox"/> Concentration (if available) % | |
| Physical form: <input type="checkbox"/> Gas <input type="checkbox"/> Liquid <input type="checkbox"/> Solid <input type="checkbox"/> Unknown | | | |
| Actual use: <input type="checkbox"/> Insecticide <input type="checkbox"/> Herbicide <input type="checkbox"/> Tick control <input type="checkbox"/> Unknown | | | |
| <input type="checkbox"/> Rodenticide <input type="checkbox"/> Fungicide <input type="checkbox"/> Other (specify) | | | |
| 9. CHEMICAL TYPE (check one or more if relevant) | | | |
| <input type="checkbox"/> Organophosphorus | <input type="checkbox"/> Thiocarbamate | <input type="checkbox"/> Dinitrophenol deriv. | <input type="checkbox"/> Fluoroacetate <input type="checkbox"/> Unknown |
| <input type="checkbox"/> Carbamate | <input type="checkbox"/> Coumarin | <input type="checkbox"/> Organomercurial | <input type="checkbox"/> Other (specify) |
| <input type="checkbox"/> Organochlorine | <input type="checkbox"/> Dipyridyl | <input type="checkbox"/> Phosphide | <input type="checkbox"/> Specific chemical: |
| <input type="checkbox"/> Pyrethroid | <input type="checkbox"/> Phenoxyacid | <input type="checkbox"/> Arsenical | |
| 10. MANAGEMENT | | | |
| Treatment given: <input type="checkbox"/> Yes <input type="checkbox"/> No <input type="checkbox"/> Unknown | | <input type="checkbox"/> Referred to other hospital | |
| Hospitalisation: <input type="checkbox"/> Yes <input type="checkbox"/> No <input type="checkbox"/> Unknown | | If yes, days in hospital Days in ICU | |
| 11. SEVERITY GRADING | | | |
| Effects: <input type="checkbox"/> Local <input type="checkbox"/> Systemic <input type="checkbox"/> Both | | PSS: <input type="checkbox"/> None <input type="checkbox"/> Minor <input type="checkbox"/> Moderate <input type="checkbox"/> Severe | |
| 12. OUTCOME | | | |
| <input type="checkbox"/> Recovery <input type="checkbox"/> Recovery with sequelae <input type="checkbox"/> Death related <input type="checkbox"/> Death unrelated <input type="checkbox"/> Unknown | | | |
| 13. COMMENTS (stating section; continue overleaf if necessary) | | | |
| | | | |

Fig. 1 Pesticide exposure record.

in SEA countries, the factors taken into consideration for selecting institutions were:

- Number of patients treated/managed during a given period.
- Accessibility/ease of travel of the population served by the facility.
- Cooperation of the medical personnel.
- Quality of the medical records.

Prospective and/or retrospective study

Although data collection should be prospective, the project allows for retrospective data collection if considered feasible, valuable, or necessary.

Time frame

The prospective studies are performed for a minimum of 12 months, subject to availability of resources (human and financial).

Management and Composition of the Study Team

The IPCS is responsible for the international coordination of the project, with the support of a selected advisory group of experts and representatives of the Regional and Country Offices of the WHO.

In each country, a Coordinator is in charge of preparing and organizing the study; obtaining clearance and administrative support; coordinating case data collection, data entry, and analysis; providing the Responsible Officers with information on pesticides; checking the product composition and use; guaranteeing the quality of the data collected and its interpretation; and preparing the reports.

The Coordinators are also responsible for implementing the study in health institutions in a country, receive and disburse funds, train professionals involved in the study, prepare reports, and coordinate activities with other agencies assisting and/or involved in the study.

The Responsible Officer(s) are in charge of collecting data, completing the PER and providing any other information required for study under the supervision of the Project Coordinator.

Other personnel involved in the country projects include medical records officer, data entry officer, administrative personnel, and epidemiologist/statistician.

STUDY IMPLEMENTATION

A guidance document is available for project participants, where the objectives of the study, preparatory

activities, study design, coordination, and implementation are described. Instructions on the preparation of country budget, project proposals, and deliverables of the study are provided, together with definitions that ensure a controlled terminology.^[3]

The IPCS provides guidance and technical support to Project Coordinators in order to ensure the harmonized data collection and its analysis. The Project Coordinators are responsible for training the personnel performing the study. Data entry and analysis is done at local level, and the Project Coordinators in each country facilitate the training of those responsible for data entry and analysis.

Meetings

Project Coordinators meet at least once a year to discuss the project activities with colleagues, the WHO country representation, the IPCS, and other relevant agencies. Local meetings are held on a regular basis, according to the needs (e.g., once a month), in coordination with Environmental Health Offices in the local WHO representations.

Data Collection

Prospective study: Data collection for the prospective study start on a specified date, from the moment the project is approved, participants selected, the health care facility identified, and the relevant health personnel briefed of the objectives and the plan of action of the study.

Retrospective study: Data collection for the retrospective study can be started at any time, transferring information from existing clinical records into the PER.

Other data: A minimum set of demographic and other indicators from each country is collected by the Project Coordinator in order to facilitate the analysis and interpretation of results. This information includes population served by the health care facility, gender/age distribution, access to health care facilities, socio-cultural characteristics of the population, migration of population during agricultural seasons, literacy level, cultural and social aspects, characteristics of the population, type and quality of pesticide equipment, patterns of pesticide use, etc. Forensic data (number of deaths due to poisoning) and health indicators of the country will also be collected from the available reference sources.

Data entry

Computer-trained personnel enter the data from the PER into the Access software system provided by the

Table 1 Results and conclusions of stage 1 studies

| Country | Duration | Participation | Number of cases |
|-----------|----------|-------------------------------|-----------------|
| India | 1 yr | 10 hospitals | 1531 |
| Indonesia | 6 mo | 7 hospitals, 1 hr office | 126 |
| Nepal | 6 mo | 4 hospitals, 1 hr institution | 258 |
| Thailand | 3 mo | 10 hospitals | 130 |

IPCS, prepared originally by the Canadian Centre for Occupational Health and Safety, Canada, and adapted to the needs of this project at the IPCS.

Data analysis

Analysis of data is done at two levels: 1) in the country, to cater for local needs; 2) centrally (WHO/IPCS), to study the pooled global data.

EXPECTED OUTPUTS

The expected outputs of the project include:

- Database on pesticide product composition.
- Report on "Health Effects of Pesticides."
- Annual reports on human pesticide exposures and their characteristics.
- Establishment of an international mechanism for toxico-vigilance and surveillance systems for pesticide poisonings.
- Identification of hazardous pesticide formulations within countries.
- Prevention of pesticide poisonings through public awareness and prevention campaigns.
- Recommendations for action at the health care level (and others, if relevant).

ACTIVITIES UNDERTAKEN

In 1992, the IPCS initiated consultations with experts in the field of pesticide poisoning. The purpose was

to develop a project to collect data on pesticide poisoning on an international basis in order to establish a sound evidence base regarding the global incidence and severity of pesticide poisoning. Standard formats were designed for collecting relevant information on cases of poisoning, and a pilot study was undertaken in three countries to test both a simple and a more elaborate data collection format. Countries were selected based on three main criteria: an agriculture-based economy, a reasonably developed product registration system, and an infrastructure for data collection and analysis. The countries selected were India, Sri Lanka, and Uruguay, which had expressed interest in implementing the project.

The tools developed (formats, guidelines, strategy, and methodology) were assessed, discussed, and improved on the basis of experience gained and data collected through this initial exercise. The material prepared was presented to representatives of countries of the WHO South East Asia Regional Office (SEARO), the Western Pacific Regional Office (WPRO), and the Americas Regional Office (AMRO) at regional workshops held in India, Singapore, and Uruguay in 1999, 2000, and 2001, respectively. Harmonized case data collection using the proposed methodology is now being implemented in selected areas from countries in those regions; other regions of WHO have been invited to participate.

Regional Activities—Trial Implementation Phase

Four countries in SEA Region, namely, India, Indonesia, Nepal, and Thailand, have completed the Trial Implementation Phase of the project. Although the coverage and duration of this trial phase differed between the countries, data were collected using a harmonized PER format, medical staff were instructed on the collection of information, on the diagnosis and treatment of cases of pesticide exposure, and on the use of the PSS. Guidance was given on developing a pesticide product register.

Some results and conclusions of stage 1 (trial) studies, period covered, number of cases, and circumstances of exposure are given in Tables 1 and 2.

Table 2 First-stage studies—Circumstances of exposure

| Country | Intentional exposure | Accidental exposure | Occupational exposure |
|-----------|----------------------|---------------------|-----------------------|
| India | 1304 (85.02%) | 72 (4.70%) | 83 (5.42%) |
| Indonesia | 54 (44.4%) | 20 (15.9%) | 47 (31.7%) |
| Nepal | 236 (91.5%) | 3 (1.16%) | 16 (6.2%) |
| Thailand | 80 (61.5%) | 10 (7.7%) | 37 (28.5%) |

CONCLUSIONS

This trial phase has confirmed that pesticide poisoning is a public health problem of importance in the South East Asia Region. The data demonstrated the magnitude of the problem due to intentional poisoning^[4] but did not appear to reflect the situation concerning occupational and accidental exposures. It was also recognized that population-based studies are required in order to collect information about cases which are not in the hospital records. The second stage of the study will include such studies, and work is being carried out in this direction.

It is foreseen that the results of the study will demonstrate not only the magnitude of the pesticide poisoning and its costs to the health services and to the society, but also issues such as the quality of clinical records and clinical services, and delivery of clinical care. The results will give an indication as how each country should plan a surveillance mechanism for pesticide poisoning as well as prevention activities.

It is also expected that information, education, and communication programs in the community would be designed according to the outcomes in individual countries.

REFERENCES

1. World Health Organization. Epidemiology of pesticide poisoning. In *Harmonized Collection of Data on Human Pesticide Exposures*; Geneva, October 2000; IPCS/EPP/00.1.
2. Persson, H.E.; Sjoberg, G.K.; Haines, J.A.; Pronczuk de Garbino, J. Poisoning severity score. Grading of acute poisoning. *J. Toxicol., Clin. Toxicol.* **1998**, *36* (3), 205–213.
3. <http://www.intox.org/firstpage.htm>.
4. Report on Establishment of Harmonized Pesticide Poisoning Database in India. Division of Medical Toxicology and Risk Assessment. Central Insecticides Laboratory. Directorate of Plant Protection, Quarantine & Storage. Ministry of Agriculture, Government of India. 2001.

Invasive Insects as Major Pests in the United States

E. Richard Hoebeke

Department of Entomology, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

The continental United States has been and remains particularly prone to exotic pest introductions. The majority of our pests originally came along with crops and livestock introduced by the early explorers and colonists. In fact, about 99% of the cultivated areas of North America is planted with introduced crops, thus making all of our croplands vulnerable to introduced pests. With these introductions came some of the insects, weeds, and other pests that affected these crops and animals in the countries from which they originated.^[1]

The rapid expansion of agriculture and commerce in North America has brought about the often accidental and unintentional introduction of many non-native species. Not all newly introduced species, however, become pestiferous; in fact, the majority cause little or no noticeable crop or environmental damage once they become established. But occasionally, a species can spread unimpeded and becomes invasive, capable of causing great economic and ecological damage to U.S. agriculture and other natural resources.

During the past 500 years, well over 2000 species of insects of foreign origin have established free-living populations in the continental United States.^[2] In addition, it is estimated that over 2500 exotic species of insects have become permanent additions to Hawaii's fauna.^[3] Although our exotic insect fauna represents a mere 2–3% of the total insect fauna known for the continental United States, the agricultural pests among this foreign assemblage account for approximately one-half.^[2,4] Calculating the full magnitude of economic costs associated with exotic species is difficult and estimates vary. During the period 1906–1991, just 43 exotic insect species caused reported losses of \$92.6 billion in harmful effects.^[2] Another study^[5] revealed that the estimated annual costs (including losses/damage and control costs) associated with a few select nonindigenous species introduced into the United States amounted to at least \$20 billion.

INVASION PHENOMENON

Since the early 1900s, there has been a continuous stream of new organisms being transported into the

United States from overseas. The rate of establishment of new exotic species has been relatively stable since about 1920, with approximately 9–12 introductions each year in the continental United States despite the deterrent effects of quarantine programs.^[4] On average, a pest of major importance has been discovered in the United States about every 3–4 years, but the pace of these pest introductions (and their subsequent detection) appears to be quickening (see Table 1).

In protecting the United States from harmful invasive species, the U.S. Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine (USDA-APHIS-PPQ) is responsible and legally mandated for excluding and managing invasive species that can potentially affect plant and animal health, either directly or indirectly. Through its quarantine and survey activities, the APHIS protects not only agriculture but also forest, rangeland, and wetland ecosystems. Unfortunately, in spite of all the safeguards in place, this exclusionary system is overwhelmed by the sheer volume of goods and commodities entering our borders by ship and by air. Past and recent breaches of the USDA-APHIS-PPQ safeguarding system have occurred, which have led to the entry of various dangerous, invasive species into the United States. And many more will likely occur in the coming decades.

A GLOBAL MARKETPLACE

International travel and the globalization of trade have ultimately led to the overwhelming increase in the frequency of introductions and the number of exotic species intercepted at U.S. ports of entry. Huge increases in trade volume and an ever-expanding list of trading partners, especially those of the Pacific Rim region and Asia during the past two decades, have resulted in the unintentional introduction and establishment of numerous exotic species in the United States. Our nation's historic first lines of defense—inspection and quarantine—are now overwhelmed by the quantity of imported commodities from around the globe. The majority of exotic species enter the United States each year as contaminants of commodities. Agricultural produce, nursery stock, cut flowers, and

Table 1 Some major exotic insects introduced into the continental United States, 1980–2001

| Year detected | Pest | State (origin) |
|---------------|---|-------------------|
| 1983 | Asian cockroach, <i>Blatella asahinai</i> (Misukubo) | FL (Asia) |
| 1985 | Asian tiger mosquito, <i>Aedes albopictus</i> (Skuse) | TX (Japan) |
| 1985 | Apple ermine moth, <i>Yponomeuta malinellus</i> (Zeller) | WA (Europe, Asia) |
| 1986 | Russian wheat aphid, <i>Diuraphis noxia</i> (Mordvilko) | TX (Asia) |
| 1992 | Lily leaf beetle, <i>Lilioceris lili</i> (Scopoli) | MA (Europe) |
| 1992 | Pine shoot beetle, <i>Tomicus piniperda</i> (L.) | OH (Europe) |
| 1993 | Citrus leafminer, <i>Phyllocnistis citrella</i> (Stainton) | FL (SE Asia) |
| 1994 | Viburnum leaf beetle, <i>Pyrrhalta viburni</i> (Paykull) | ME (Europe) |
| 1994 | Red-haired pine bark beetle, <i>Hylurgus ligniperda</i> (F.) | NY (Europe) |
| 1996 | Asian long-horned beetle, <i>Anoplophora glabripennis</i> (Motschulsky) | NY (China) |
| 2000 | Soybean aphid, <i>Aphis glycines</i> (Matsumura) | WI (Asia) |
| 2001 | Citrus long-horned beetle, <i>Anoplophora chinensis</i> (Forster) | WA (China, Japan) |
| 2001 | Brown marmorated stinkbug, <i>Halyomorpha halys</i> (Stal) | PA (China, Japan) |

timber can harbor insects and a multitude of other organisms.^[6]

BY SEA OR BY AIR

The pathways by which exotic insects enter the United States are dynamic, changing with technological advances that affect commerce and with changes in the commodities that move in commerce.^[7] The majority of commodities that enter the United States today arrive either by ship or by airplane. About 80% of the world's commodities travel by ship for at least part of the journey to their consumers, and the volume of sea-borne trade is climbing steadily upward;^[8] in fact, from 1970 to 1996, maritime trade nearly doubled.

Stowaways have been a source of nonnative species in the United States since the days of the early sailing ships. During this period, dry ship ballast provided a pathway of entry for many soil-dwelling organisms. Invasive insects such as fire ants, mole crickets, vegetable weevil, and white-fringed beetles all found their way into the United States through the dumping of soil ballast at port-of-entry sites.^[4] After the Civil War, the importation of nursery stock increased dramatically and with it a substantial increase in the number of introduced plant pests, such as scales, aphids, leafhoppers, plant bugs, and some moth species.^[4,9] In the present era, ocean-going ships and international aircraft have assumed increasing importance as pathways of entry. Air traffic alone represents a quantum leap in speed, and air cargo is a rapidly expanding sector in the trade network,^[8] growing at about 7% annually. Military cargo transport also brings in harmful species, such as the Asian gypsy moth.

Today, perhaps the two most dangerous conveyances that easily transport unwanted alien insect invaders are containerized cargo and solid wood packing material (SWPM).

Containerized Cargo

A major leap in invasion potential involves bulk freight containers—those huge metal boxes that have revolutionized the freight industry during the past couple of decades.^[8] These containers are ubiquitous and they move either by ship, by rail, or by road. They offer a safe haven to anything that manages to get inside, and they can remain stacked for weeks or even months in foreign ports or railroad yards, allowing ample time for pests to enter. They are rarely cleaned between shipments, they may not be unpacked until they are hundreds of miles from their ports of entry, and, most importantly, they are difficult to inspect. For these reasons alone, freight containers have been identified as a significant pathway for the unintentional introduction of many insects, weeds, slugs, and snails of foreign origin.

Solid Wood Packing Material

The escalation in global trade and a parallel increase in the use of solid wood packing material (SWPM) in the international trade industry have, together, combined to create one of the greatest and most perilous threats yet to the long-term health of North American urban and native forests. SWPM includes wood dunnage (sometimes with bark attached), boxes, crating, pallets, spools, and large-dimensional blocks and skids that are used in stabilizing imported cargoes in the holds

Table 2 A select sampling of injurious insects not yet known to occur in the United States but with high potential for introduction

| Pest | Country of origin | Injurious to |
|---|---------------------------------|---------------------------------------|
| European chestnut weevil, <i>Curculio elephas</i> (Gyllenhal) | SE Europe, Algeria | Chestnuts, acorns |
| Apple blossom weevil, <i>Anthonomus pomorum</i> (L.) | Europe, Asia | Pomaceous fruit |
| Melon fly, <i>Dacus cucurbitae</i> (Coquillett) | E Africa, SE Asia, N India | Cucumber, tomato, cucurbits preferred |
| European cherry fruit fly, <i>Rhagoletis cerasi</i> (L.) | Continental Europe, Asia Minor | Cherries, plums |
| Large pine weevil, <i>Hylobius abietis</i> (L.) | Europe, N Asia | All conifers, many hardwoods |
| Nun moth, <i>Lymantria monacha</i> (L.) | Europe, Asia | Conifers, deciduous trees |
| Pine sawfly, <i>Diprion pini</i> (L.) | Europe, Siberia, N Africa | Conifers |
| Senn (or Sunn) pest, <i>Eurygaster integriceps</i> (Puton) | E Europe, Middle East, N Africa | Grain crops |
| Silver-Y moth, <i>Autographa gamma</i> (L.) | Europe, Asia, India, N Africa | Various crops, grasses, cereals |
| European spruce bark beetle, <i>Ips typographus</i> (L.) | Europe, Asia | Conifers, mostly spruce |

(From Ref. ^[13].)

of ships and in bulk freight containers. The recent detection of the Asian long-horned beetle in New York in 1996 and in Illinois in 1998 has emphasized the importance of solid wood packing material as a conveyance for the unintentional importation of exotic forest pests.^[7] The pine shoot beetle, *Tomicus piniperda* (L.), was discovered near Cleveland, OH, in 1992, infesting shoots of white pine in a Christmas tree plantation; subsequent surveys for the pest in the following years have detected it in at least 12 states in the Great Lakes region and surrounding states.^[10] This European invader is believed to have escaped from wood dunnage discarded at various Great Lakes port-of-entry sites sometime during the mid or late 1980s. The detection of a severe infestation of red spruce in Halifax, Nova Scotia, Canada, in 2000 by the brown spruce longhorn beetle, *Tetropium fuscum* (F.),^[11] led to the removal and the destruction of nearly 6000 mature trees in a local park setting of that maritime city. This Eurasian beetle probably arrived in wood crating or dunnage offloaded from freight containers of a nearby marine container terminal. Because of its close proximity, it poses a serious threat to the coniferous forests of the northeastern United States.

A DANGEROUS RESERVOIR AWAITS

There remains a substantial reservoir of foreign species of insects that are potentially injurious to American agriculture and forestry, awaiting transportation to North America. Many of these are fully expected to become pests upon their arrival and establishment; a mere sampling of some of these potential crop pests is listed in Table 2. As many as 6000 species of insects (and mites) are known pests in various foreign regions that serve as serious threats to U.S. agriculture. In a

report called *The Emigrant Pests*, a U.S. task force^[12] examined some of these and concluded that nearly 600 may be regarded as high-risk. If they become established, these exotic pests are expected to produce a wide range of economic impacts on U.S. agriculture. In fact, it has been estimated that as little as 2% of these may produce impacts from \$400 million to \$4 billion in damage or crop losses, whereas 75% of them may produce impacts of less than \$4 million.^[12]

Exclusion is obviously desirable; however, chances are reasonably high that some of these will become established in any given year. Alarmingly, it has been suggested^[12] that there is no objective evidence that U.S. quarantine actions are having any significant impact on this steady flow.

REFERENCES

1. Council for Agricultural Science and Technology. *Pests of Plants and Animals: Their Introduction and Spread*. Council for Agricultural Science and Technology: II. Series Report, No. 112; 1987; 36 pp.
2. U.S. Congress, Office of Technology Assessment. *Harmful Non-indigenous Species in the United States (OTA0F-565)*. U.S. Government Printing Office: Washington, DC, 1993; 391 pp.
3. Funasaki, G.Y.; Lai, P.-Y.; Nakahara, L.M.; Beardsley, J.W.; Ota, A.K. A review of biological control introductions in Hawaii: 1890 to 1985. *Proc. Hawaii. Entomol. Soc.* **1988**, *28*, 105–160.
4. Sailer, R.I. History of insect introductions. In *Exotic Plant Pests and North American Agriculture*; Wilson, C.L., Graham, C.L., Eds.; Academic Press: New York, 1983; 15–63.
5. Pimentel, D.; Lach, L.; Zuniga, R.; Morrison, D. Environmental and economic costs of nonindigenous species in the United States. *Bioscience* **2000**, *50* (1), 53–65.

6. <http://www.aphis.usda.gov/oa/pubs/invasive.html> (accessed December 2001).
7. Wheeler, A.G., Jr.; Hoebeke, E.R. A history of adventive insects in North America: their pathways of entry and programs for their detection. In *Detecting and Monitoring Invasive Species*, Proceedings of the USDA, APHIS Plant Health Conference 2000, Raleigh, NC, Oct 24–25, 2000; 2001; 3–15.
8. Bright, C. *Life Out of Bounds: Bioinvasion in a Borderless World*; W. W. Norton: New York, 1998; 287 pp.
9. Sailer, R.I. Our immigrant fauna. *Bull. Entomol. Soc. Am.* **1978**, *24*, 3–11.
10. Haack, R.A.; Poland, T.M.; Petrice, T.R.; Gennrich, M.A. Range expansion of *Tomicus piniperda* since 1992 and notes on other new exotics. *News. Michigan Entomol. Soc.* **2000**, *45* (3–4), 9–10.
11. Smith, G.; Hurley, J.E. First North American record of the Palearctic species *Tetropium fuscum* (Fabricius) (Coleoptera: Cerambycidae). *Coleopt. Bull.* **2000**, *54* (4), 540.
12. McGregor, R.C. The emigrant pests. In *A Mimeographed Report to Dr. Francis J. Mulhern, Administrator*; U.S. Department of Agriculture: Washington, DC, 1973.
13. Lattin, J.D.; Oman, P. Where are the exotic insect threats? In *Exotic Plant Pests and North American Agriculture*; Wilson, C.L., Graham, C.L., Eds.; Academic Press: New York, 1983; 93–137.

IPM Farmer Field School

John Pontius

*Training Specialist, The FAO Programme for Community IPM in Asia,
Jakarta, Indonesia*

INTRODUCTION

The IPM Farmer Field School (FFS) provides farmers with the education they need to sustainably manage their agro-ecosystems.^[1] The first FFS, which focused on irrigated rice, was conducted in Indonesia during the rainy season of 1989–1990 as part of the Indonesian National IPM Program. The FFS became the educational approach used in national IPM programs supported by the FAO Inter-Country Program for Integrated Pest Control in Rice in South and Southeast Asia. By mid-1990s, FFS were being conducted throughout Asia and Southeast Asia in rice as well as vegetable and estate crops. By the late 1990s, the IPM Field School approach was being applied in regions outside of Asia, most notably Africa. The FFS provides small farmers with practical experience in ecology and agro-ecosystem analysis.^[2] Farmers acquire the analytical skills that they need to practice IPM and create solutions to the agro-ecosystem problems that they face.^[3]

FOUR PRINCIPLES

The IPM FFS is based upon four principles.^[4] The principles provide a guide to what farmers should be able to do because of participation in an FFS. These principles are:

- Grow a healthy crop
- Conserve natural enemies
- Conduct regular field observations
- Become IPM experts

The first principle means that FFS participants will need to be able to apply good agronomic practices and understand plant biology. This should help alumni to optimize their yields as well as grow plants that can withstand disease and pest infestations. The second principle implies that FFS alumni will reduce their use of insecticides. To do this, FFS participants will need to understand insect population dynamics and field ecology. The third principle asserts that IPM requires of farmers the ability to regularly observe, analyze, and take informed decisions based on the

conditions of their agro-ecosystems. The fourth principle posits that because of local specificity, farmers are better positioned to be taking the decisions relevant to their fields than agriculture specialists in a distant city. Hence, FFS alumni should be able to apply IPM in their fields and also be able to help others do so.^[5]

THE IPM FIELD SCHOOL APPROACH

The FFS approach features several departures from earlier IPM farmer training and agriculture extension approaches.^[6,7] Included among these innovations are field-based, season-long learning for farmers, field experiments, a focus on plant biology and agronomic issues, a new method for agro-ecosystem analysis, the inclusion of human dynamics activities, and a learning approach that stresses participatory discovery learning.^[8] The FFS experience provides farmers with an educational foundation upon which they can further build to enhance their abilities to employ not only IPM, but also other knowledge intensive forms of agriculture.^[9]

The following is a list of the basic characteristics of an IPM Farmer Field School.

- The IPM Field School is field based and extends over a full cropping season.
- FFS meetings are conducted on a weekly or biweekly basis depending on the length of a given crop's cycle and the rapidity of change in agro-ecosystem factors (e.g., for rice weekly, for cacao biweekly).
- The primary learning material at an FFS is the field; participants generate other learning materials based on the field.
- The FFS meeting place is close to the learning field, often in a farmer's home and sometimes beneath a convenient tree.
- FFS educational methods are experiential, participatory, and learner centered.
- Each FFS meeting includes at least three activities: the agro-ecosystem analysis, a "special topic," and a group dynamics activity.
- In every FFS, participants conduct a study comparing IPM with non-IPM treated plots.

- An FFS often includes several additional field studies depending on local field problems.
- Between 25 and 30 farmers participate in an FFS. In all FFS activities, participants learn together in small groups of five.

All learning activities in an FFS are based on the “experiential learning cycle.”^[10] In a given activity, the cycle begins with learners going through a concrete experience that provides the “data” or material for learning. Next, learners are given a chance to reflect on or analyze the experience. Based on their analysis, learners next generalize or develop a hypothesis. This hypothesis is then tested via experimentation and the cycle, essentially, starts anew.

Typically, an FFS meeting lasts about four hours and begins in the early morning when there is still a lot of insect activity in the field. Three basic activities comprise an FFS meeting, the agro-ecosystem analysis, a special topic activity, and a group dynamics activity.^[11] The agro-ecosystem analysis consists of three stages. First, in their five-member teams, participants enter the FFS learning fields to *observe* general field conditions, sample plants, collect insects, make notes, and gather live specimens. The field provides all of the basic learning materials and subject matter for the FFS. Next, each team *analyzes* their field samples and notes by creating a visual analytical tool known as the agro-ecosystem drawing. This tool is made up of key ecosystem factors such as pest/predator densities, plant health, field conditions, weather, and current management treatment. The output of the analysis is a field management decision. Finally, after the analytical session, a member of each small group *presents* his or her group’s analysis and decisions to the rest of the members of the FFS. The presenter and his or her group then defend their analysis in open discussion. “What if...?” or problem-posing questions are used by the facilitator to further hone the analytical skills of participants during this discussion.^[12]

The special topic activity is linked to the stage of growth of the crop and specific local issues. Special topics are selected from a large “menu” of potential topics that are mastered by FFS facilitators during their training. The topics selected for an FFS are based on local conditions. Special Topic activities include crop physiology, health and safety, food webs, field ecology, economic analysis, water management, and fertilizer use. Most, but not all, of these exercises require being in the field.

Group dynamics activities focus on problem solving, communication, leadership, and team building. These activities typically use simulation exercises to create situations in which participants work at resolving a problem. After the simulation, FFS participants

compare and analyze their solutions, the processes they went through in arriving at their solutions, and how what they learned via the group dynamics activity can be related to “real” life.

TYPICAL FFS STUDIES

All FFS rely on studies or experiments to help farmers learn. Special topic studies such as the insect “zoo” and field studies are two examples of FFS studies. The insect zoo is essentially an exclusion study in which an enclosure is built to contain specific insects together with a specific plant such as a rice plant and exclude all other insects. The insect zoo in a rice IPM FFS consists of a rice plant that is placed in a pot and enclosed either with clear plastic or very fine netting. Insect zoo studies should be conducted as part of any FFS. The insect zoo study can focus on locally identified pest problems, but it is often used in connection with any of several possible general topics including pest/predator relationships, insect life cycles, and plant/insect relationships. Often, the insect zoo is used to help learners discover the predatory capacities of natural enemies. In general, the insect zoo helps FFS participants to increase their understanding of ecological principles in their agro-ecosystems.

A fairly simple rice IPM FFS insect zoo study would be to examine the capacity of a wolf spider to consume Brown Plant hoppers (BPH). The spider and a number of BPH—the number of BPH should be sufficient to provide the spider a couple days of hunting, maybe 40 adults—are placed together in the zoo and the zoo is sealed. FFS participants are then asked to observe the insect zoo over the week between FFS meetings and take note of what happens. The following week at the FFS, the participants would analyze what happened in their studies and discuss the roles of wolf spiders and spiders in general in the rice agro-ecosystem.

In contrast to special topic studies, which are usually short exercises, a field study usually continues for several weeks up to the entire season of the FFS. One example of an FFS field study conducted in every FFS is the *comparison study* of IPM and non-IPM treated field plots. The comparison plots could be further divided into subplots to conduct additional studies, often known as *supporting studies*. Typically, these studies concern agronomic or ecological issues and help farmers to learn the process of doing applied field research.^[13] Analysis of either a comparison study or a supporting study requires taking a yield cut to compare the number of tillers, the number and weight of grains, and yields among the various treatments in the study.

An example of a common supporting field study at a rice FFS is to demonstrate the capacity of a plant to compensate for damaged or lost tillers. In this study, plants in “study blocks” within one of the larger comparison study fields would have their tillers cut up to about 55 days after transplanting (DAT). The study could be as simple as cutting 20% of the leaves in 1 m² blocks at 14, 30, and 55 DAT. During the FFS, participants would make weekly observations of the growth of the treated plants and take note of the numbers of tillers in these plants.

THE FUTURE

By the late 1990s, the FFS was being used as the starting point for building community-based IPM programs. By year 2000, over 30,000 farmers in Asia and Southeast Asia had been trained to conduct FFS. These farmer IPM trainers are the leadership core for village IPM programs across Asia that have led to, among other things: FFS becoming part of formal schools’ curricula, communities producing and marketing pesticide-free rice, a national association of IPM farmers in Indonesia, farmer-led research and learning centers, local funding for FFS alumni activities across the region, and countless alumni advocacy activities aimed at improving, among other things, government policy to support their village IPM movements.

REFERENCES

1. Dilts, R. Training: re-schooling society? *Prisma* **1985**, 38, 78–90 (December).
2. Pretty, J. *Regenerating Agriculture*; Earthscan Publications, Ltd.: London, 1995.
3. Mezirow, J. A critical theory of adult education. *Adult Educ.* **1980**, 31, 1–23 (Fall).
4. Indonesian IPM Program. *IPM By Farmers*; Ministry of Agriculture: Jakarta.
5. Freire, P. *Pedagogy of the Oppressed*; The Seabury Press: New York, 1970.
6. Roling, N.G., Wagemakers, M.A.E., Eds.; *Facilitating Sustainable Agriculture*; Cambridge University Press: Cambridge, 1998.
7. Mangan, J.; Mangan, M.S. A comparison of two IPM training strategies in China: the importance of concepts of the rice ecosystem for sustainable insect pest management. *Agric. Human Values* **1998**, 15, 209–221.
8. Knowles, M. *The Adult Learner: A Neglected Species*, 2nd Ed.; Gulf Publishing Company: Houston.
9. Habermas, J. *Knowledge and Human Interests*; Beacon Press: Boston, 1971.
10. Kolb, D.A. *Experiential Learning*; Prentice-Hall: Englewood Cliffs, NJ, 1984.
11. FAO Technical Assistance Team. *Community IPM: Six Cases from Indonesia*; FAO-Technical Assistance Team; Indonesian National IPM Program: Jakarta, 1998.
12. Rogers, C. *Freedom to Learn*; Merrill: Columbus, OH, 1969.
13. van den Berg, H.; Lestari, A.S. Improving local cultivation of soybean in Indonesia through farmers’ experiments. *Exp. Agric.* **2000**, 36, 1–11.

Irradiation

Hitoshi Ito

Takasaki Radiation Chemistry Research Establishment, Japan Atomic Energy Research Institute, Takasaki, Gunma, Japan

INTRODUCTION

Irradiation has been shown to be an effective pest control method for stored foods. Radiation can be used for insect disinfestation and microbial control. The type of radiation used during processing is limited to ionizing radiation from high-energy gamma rays, X-rays, and accelerated electrons without making materials radioactive. Gamma rays and X-rays have high penetration capacity compared with electron beams.

Radiation is an effective pest control method for insects and mites in cereals, dried fishes, and fresh fruits, and a good alternative to methyl bromide fumigation.

The shelf life of many fruits and vegetables, meat, poultry, fish, and seafood can be prolonged by radiation combined with other methods such as refrigeration or packaging. Reduction or inactivation of microorganisms such as putrefactive bacteria, pathogenic bacteria, and fungi in food can be achieved by radiation treatment without changing sensory quality and nutrition.

IRRADIATION OF FOODS

Irradiation has been shown to be an effective pest control method for insect disinfestation and microbiological control for stored foods. The type of radiation used for pest management in foods is limited to radiation from high energy of gamma rays and X-rays having a maximum energy of 5 MeV or accelerated electrons having a maximum energy of 10 MeV. The process involving this radiation dose cannot make food radioactive. Gamma rays and X-rays have high penetration capacity compared with electron beams. These kinds of radiation are referred to as ionizing radiation because their energy is high enough to dislodge electrons from atoms and molecules and produce free radicals in foods. Radiolytic products in foods are mainly formed by indirect action of water through hydroxyl radical ($\cdot\text{OH}$), hydrated radical (e_{aq}^-), and hydrogen radical ($\cdot\text{H}$).

The amount of radiolytic products in irradiated food is below 30 mg per 1 kg at 1 kGy, and mainly consist of sugars, amino acids, and small amounts of

hydrocarbons, alcohols, aldehydes, fatty acids, carbon dioxide, etc.—which are naturally present in foods or are formed by thermal cooking. Several hundred toxicological studies in many countries have concluded that irradiation does not produce any toxic products in foods.^[1] The effect of irradiation also does not lead to loss of nutritional value in foods.

Radiation dose is the quantity of radiation energy absorbed by food as it passes through the radiation field. An absorbed dose unit is called Gy (gray; 1 J/kg). A dose of 10 kGy (10,000 Gy) is equivalent to increasing the temperature of water by 2.4°C.

INSECT AND MITE DISINFESTATION

The main problem encountered in the preservation of grains and dried fishes is insect and mite infestation. Most of such pests—beetles, moths, weevils, and mites—cause extensive damage to stored products. Fruit flies are easily distributed by trade through fresh fruits and vegetables and have accounted for great losses in agriculture.

Irradiation has been shown to be an effective pest control method to stored dry products and fresh fruits.^[2] The dosage required for insect and mite control is reasonably low—in the order of 1 kGy or less—and which does not cause undesirable changes in flavor, test, color, or texture. To inflict immediate lethality against insects and mites, doses in the range of 3–5 kGy would be required. A dose of 0.2–0.5 kGy would be sufficient if the goal is lethality within a few weeks and sterility of living insects in grains, as shown in Fig. 1. Usually, weevils, beetles, and fruit flies are less resistant to radiation than mites or moths. This sensitivity of insects to irradiation depends on the growth stage as well as species. Eggs and pupae are more sensitive than larvae or adults. Any progeny of insects or mites would be sterile as a result of genetic damage.

Insects and mites in cereal grains such as wheat, rice, and spices, or dried fishes can be controlled by irradiation treatment involving a range 0.2–0.5 kGy. However, proper packaging or interception is required for irradiated products to prevent insect reinfestation. Fruit fly infestation in fresh fruits and vegetables can be controlled with 0.15–0.3 kGy. Radiation

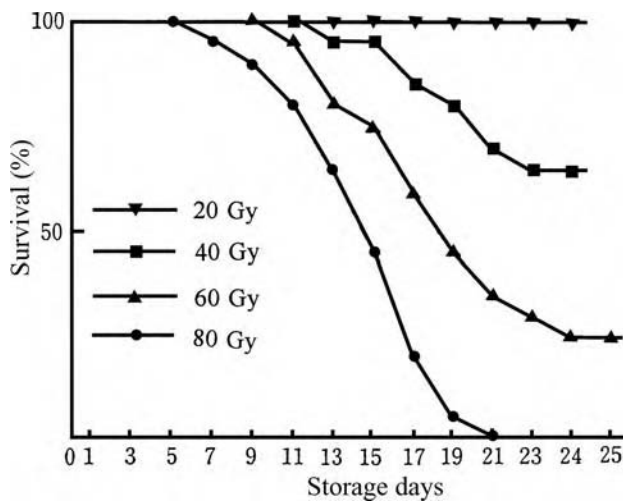


Fig. 1 Survival of adult maize-weevil during storage at 30°C, 70–80% R.H. in rice after irradiation.

disinfestation can facilitate trade in fresh fruits, such as citrus, mangoes, and papayas, which often cause problems that may require quarantine handling. Irradiation is a good alternative to methyl bromide, the most widely used fumigant for pest control.

CONTROL OF MICROORGANISMS

The main problem of stored foods such as meat, poultry, fish, and seafood is putrefaction by microorganisms. The shelf life of these foods can be prolonged by treatment with combinations of low-dose irradiation and refrigeration that do not alter sensory qualities. Many spoilage bacteria and yeasts are relatively sensitive to radiation except spore-forming bacteria. For example, a dose of at least 3 kGy applied to fresh chicken meat will be enough to eliminate *Salmonella*, *Escherichia coli* O157, and *Listeria*, and will also kill many, but not all, spoilage bacteria,^[3] as shown in Figs. 2, and 3. Compared with putrefactive bacteria, many pathogenic bacteria such as *E. coli* O157:H7, *Salmonella*, *Campylobacter jejuni*, *Listeria monocytogenes*, *Vibrio* are sensitive to radiation and can be eliminated at 3–6 kGy irradiation under appropriate production conditions.^[2,4]

Mold growth in grains, spices, dried fishes, or animal feeds during storage at high humidity have likewise caused huge problems due to decrease in quality or nutritional value and production of micotoxins. The growth of molds such as *Aspergillus* or *Penicillium* can be controlled at doses of below 5 kGy.^[5] In well-dried grains or spices, a dose for insect disinfestation can suppress the growth of molds during storage.

Strawberries are frequently spoiled by *Botrytis* molds. Treatment with a dose of 2–3 kGy of gamma

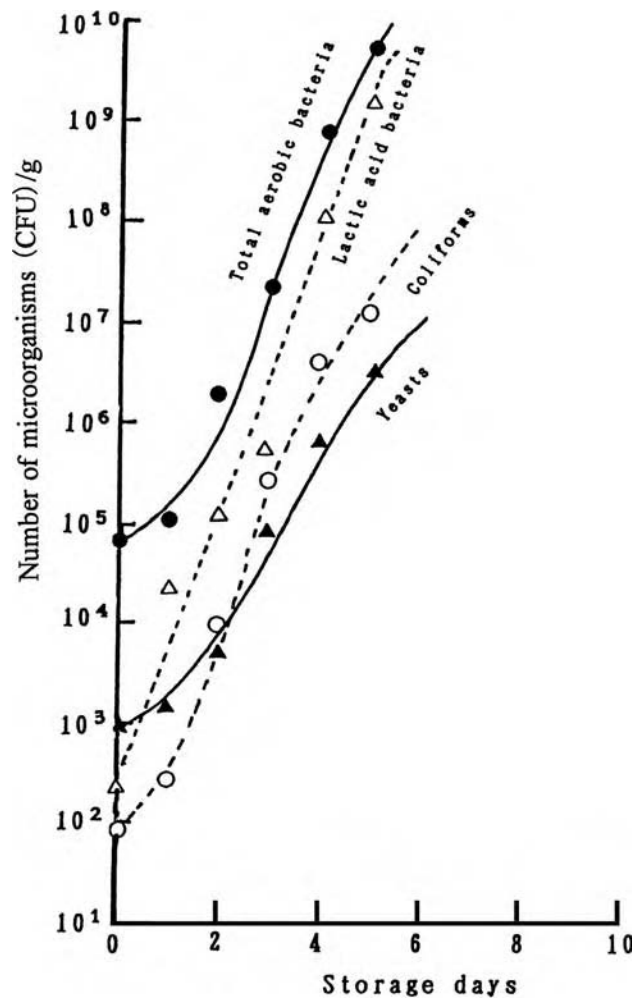


Fig. 2 Main microbial growth pattern of non-irradiated chicken meat at 10°C.

rays or X-rays followed by storage below 10°C can prolong a shelf life up to 14 days. Citrus fruits are spoiled by some kinds of mold like *Penicillium*. Treatment with a dose of 1.5–2 kGy of low-energy electron beams at 0.5 MeV on the skin surface followed by storage below 10°C can prolong a shelf life up to 2–3 months.^[6] Treatment combining a low dose at 1 kGy and heating at 40–50°C can inactivate many phytopathogenic fungi on fruits and vegetables. However, not all fruits and vegetables are suitable for irradiation because undesirable changes in color or texture, or both at doses exceeding 1 kGy, can affect the limits of their acceptability.

At high doses of irradiation, meat and poultry, which are preheated to inactivate enzymes at medium cooking of 75°C, can be commercially sterilized at –40°C, like what is being done in canning by thermal sterilization. Radiation can sterilize *Clostridium botulinum* type A or E at 25–50 kGy.^[2] It is important that radiation-sterilized meat and poultry retain much

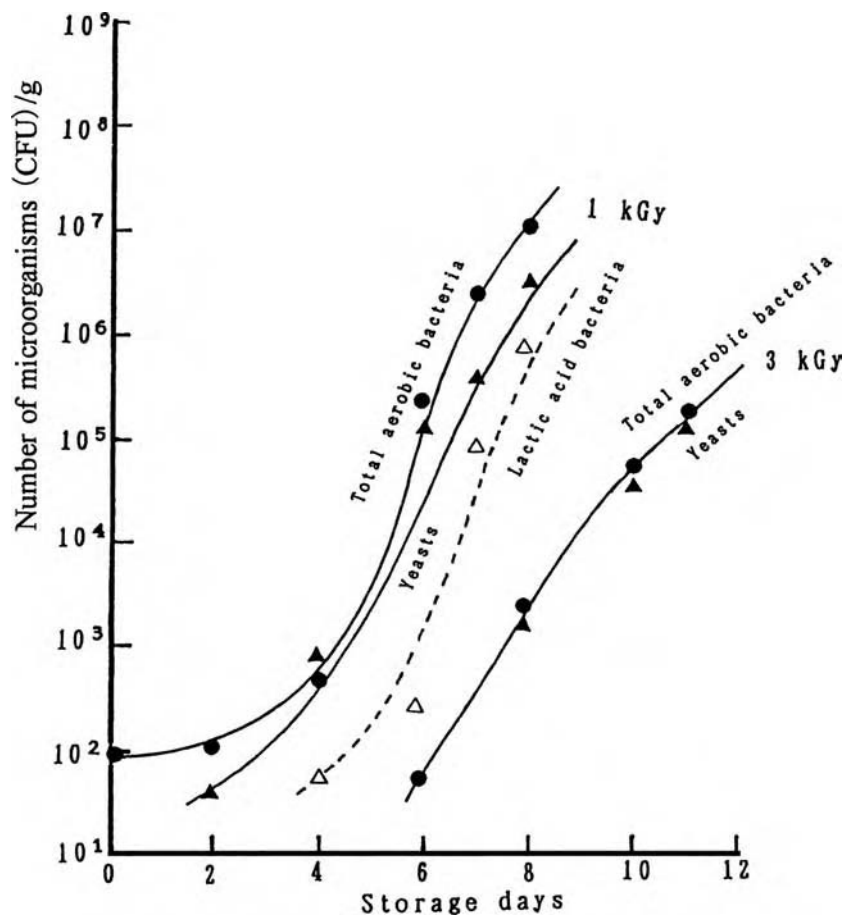


Fig. 3 Main microbial growth pattern of chicken meat irradiated at 1 and 3 kGy and stored at 10°C.

more nutrients like vitamins even at B1 and C than canning by thermal sterilization.

REFERENCES

1. Report of a Joint FAO/IAEA/WHO Study Group. In *High-dose Irradiation: Wholesomeness of Food Irradiation with Doses Above 10 kGy*; WHO Technical Report Series, World Health Organization: Geneva, 1999; Vol. 890.
2. Molins, R.A., Ed.; *Food Irradiation Principles and Applications*; J. Wiley & Sons, 2001.
3. Yutapong, P.; Bannati, D.; Ito, H. Shelf life extension of chicken meat by γ -irradiation and microflora. *Food Sci. Technol. Int.* **1996**, *2* (4), 242–245.
4. Rashid, H.O.; Ito, H.; Ishigaki, I. Distribution of pathogenic vibrios and other bacteria in imported frozen shrimps, and their decontamination by gamma-irradiation. *World J. Microbiol. Biotechnol.* **1992**, *8*, 494–499.
5. Juri, M.L.; Ito, H.; Watanabe, H.; Tamura, N. Distribution of microorganisms, and their decontamination by gamma-irradiation. *Agric. Biol. Chem.* **1986**, *50* (2), 347–355.
6. Takehisa, M.; Ito, H. Experiences of food irradiation in Japan. *Food Rev. Int.* **1986**, *2* (1), 19–44.

Landscape Ornamentals

Michael J. Raupp
Paula M. Shrewsbury

Department of Entomology, College of Life Sciences, University of Maryland, College Park,
Maryland, U.S.A.

INTRODUCTION

Landscape plants are important owing to revenues generated for their growers, the value added to real estate, and the ameliorative impact on people and urban and suburban environments. The production of ornamental plants in the United States was estimated at U.S.\$10.9 billion in 1996, with retail expenditures for floriculture and environmental horticulture at U.S.\$37.2 billion.^[1] In 2002, over 21 million households in the U.S. purchased services for landscape, lawn, and tree care.^[2] Landscape plants are estimated to add U.S.\$1.5 billion dollars annually to the value of residential real estate in the United States. Landscape plants remove pollutants from the air, contribute to personal well being, and ameliorate high temperatures in urban centers, thereby reducing energy costs. Ornamental landscape plants also serve as a refuge for a diverse array of wildlife.^[3]

RATIONALE FOR PEST MANAGEMENT IN LANDSCAPES

Pesticide use is high in managed landscapes because landowners struggle to maintain the health, beauty, and value of plants.^[4] Large and thriving landscape, lawn, and tree care industries earned more than U.S.\$16 billion in 1999 and significant portions of these revenues came from treating plants.^[1,5] Homeowners and some segments of the landscape maintenance industry employ environmentally disruptive practices such as routine applications of pesticides, the treatment of pests on sight (“see and spray”), and the use of cover sprays with broad-spectrum, residual pesticides.^[4,6,7] However, progress has been made in some segments of the landscape maintenance industries that have embraced the concepts of integrated pest management (IPM) and a related approach called plant health care (PHC).^[8]

BIOLOGICAL DIVERSITY IN MANAGED LANDSCAPES

The horticultural diversity of urban habitats is large and generally much greater than typical agricultural

systems. Frankie and Ehler^[9] reported more than 330 species of woody plants within the city limits of Austin, Texas, and a study of 26 homesites in Maryland revealed in excess of 133 species and cultivars of plants under the management of a single firm.^[10]

The number of arthropod pest species in residential landscapes is large and directly related to the number and taxa of ornamental plants.^[11] However, structurally complex landscapes house lower numbers of certain pests (mites, lace bugs, and scales) than simple ones.^[12–14]

The natural enemy community in managed landscapes and urban forests is poorly understood in terms of taxonomic diversity. The few comprehensive studies to date reveal large and complex communities of natural enemies in landscape settings.^[9,12,15–21] Factors related to the vegetational texture of the landscape habitat appear to have an important affect on the diversity and abundance of natural enemies. In some, but not all, cases, structurally complex landscapes have been shown to house a greater number and a diversity of natural enemies.^[12,14,18,19] Nonetheless, natural enemies have been suggested as one mechanism for lower pest abundance in complex landscapes than simple ones.

BIOLOGICAL DIVERSITY OF LANDSCAPES IN A MANAGEMENT CONTEXT: KEY PLANTS AND KEY PESTS

The concept of key pests is widely used in agronomic systems to define the focus for management activities. Although overall pest diversity is large in landscape systems, a rather limited number of insects and mites create the majority of problems. Data gathered from scouting programs in residential landscapes, institutional grounds, and urban forests disclosed that 10 species or functionally related groups accounted for 63–97% of the arthropod pests encountered annually.^[5] Lists of key pests in a geographical region appear relatively stable temporally but vary spatially.^[5]

Key plants provide aesthetical or functional attributes that contribute significantly to the landscape value,^[22] and are most likely to incur serious, perennial problems that dominate control practices.^[23] In

examining more than 14,000 home landscape plants, no relationship existed between the commonness of a plant taxon and its frequency of being attacked. Common genera of plants were just as likely to incur arthropod problems as rare ones. However, regardless of their relative abundance in the landscape, plants in the family Rosaceae were significantly more likely to have arthropod pests than plants in other families.^[24] The identification of key plants can assist in the design of pest-resistant landscapes and will focus the monitoring and intervention activities of IPM and PHC programs.^[5,25]

NEW APPROACHES FOR MONITORING

Monitoring, sometimes referred to as scouting, provides the pest manager with critical information on pest identification, the occurrence of pests and beneficials in time and space, the presence of susceptible life stages to target control measures, and damage estimates. Time spent monitoring landscape plants is related to building lot size, plant abundance and richness, and key plant abundance.^[10,11,25] Regular visual inspections continue to be the most widely used monitoring technique. Other monitoring techniques include pheromone traps and degree day (DD) accumulations used by environmental data loggers.^[5] Degree day accumulations have been established for many key pests.^[26]

Recently, there has been an emphasis on the use of plant flowering phenology as a correlate of insect activity.^[27–29] Plant phenological indicators (PPIs) are easy to use and thereby attractive to landscape managers, growers, and homeowners alike. One drawback of this method is that PPIs developed for one geographical zone are not necessarily accurate for other zones.^[28,29] The geographical information system (GIS) has been used to track and predict large-scale movements of migrant pests. Information on the spatial distribution and movement of pests can be obtained by combining information on the spatial structure of pest resources within the landscape with spatial modeling. Brewster, Allen, and Kopp^[30] used this approach to model the distribution of whitefly populations in Imperial Valley, California. This technology holds great potential for monitoring pest activity at several spatial scales, including the landscape level.

DECISION MAKING

Decision making in landscapes is complex and a variety of approaches have been used. Unfortunately, green industry practitioners and homeowners often rely on routine applications of pesticides, or methods

known as “see and spray,” to guide intervention.^[6,31] Olkowski^[32] was the first to propose the existence of aesthetical injury levels and to suggest their utility in landscape ornamentals. Recently, Sadof and Raupp^[33] reviewed the development of aesthetical-based decision-making guidelines in 15 systems involving ornamental plants. In general, consumers detect problems and consider action at levels of injury below 10% of the affected plant or landscape. In nurseries systems where ornamental plant values are high, low thresholds are the rule. Furthermore, relatively costly approaches, such as the use of expensive pesticides, routine applications, or the use of effective biological controls, may predominate. Educating people to tolerate greater levels of injury to plants is an important goal in landscape systems if pesticide use is to be reduced.^[33] Commercial arborists have combined surveys of client opinions with expert evaluations to guide intervention for landscape plants using an approach called the appropriate response process.^[8]

ADVANCES IN INTERVENTION TACTICS AND STRATEGIES

Biological Control

Classical biological control is the redistribution of natural enemies from the aboriginal home of an exotic pest to its new location. This approach has been successful in a variety of agricultural and urban landscapes systems. Paine et al.^[34] reviewed classical biological control programs for the ash whitefly, *Siphoninus phillyrae*, and the Eucalyptus long-horned borer, *Phoracantha semipunctata*, in California. Other successes involving classical biological control in landscapes include dramatic reductions in the gypsy moth, *Lymantria dispar*, by the fungus, *Entomophaga maimaiga*,^[35,36] and the control of obscure scale, *Melonaspis obscura* (Comstock), by the redistributed aphelinid, *Encarsia aurantii* (Howard).^[37]

Conservation biological control involves two strategies to conserve natural enemies: avoiding management practices detrimental to natural enemies such as the use of broad-spectrum pesticides, and enhancing the habitat to make it more favorable for natural enemies.^[38] We still have much to learn about the restoration of ecological function to managed landscapes before landscape design can be used as a reliable tool for enhancing and conserving natural enemies. However, structurally complex landscapes have both greater numbers of alternate prey and predators.^[19] Adding flowering plants to landscapes provides floral resources and refuge that enhance natural enemies.^[38]

The augmentation of natural enemies has been evaluated and is in limited use in nurseries and

landscapes. Shrewsbury and Raupp^[39] discuss 28 studies evaluating augmentative releases in nurseries using coccinellids, chrysopids, phytoseiids, nematodes, and fungi. Levels of control varied between 0% and 100%. In landscape systems, releases of the convergent lady beetle reduced aphid densities whereas releases of green lacewing larvae provided no control of aphids.^[40] Entomopathogenic nematodes have been used to control clearwing borers attacking several species of woody plants^[41] but did not control root weevils in planting beds.^[42]

Host Plant Resistance

Host plant resistance represents a durable, environmentally responsible approach for managing pests in landscapes. Yet, host plant resistance has not been broadly implemented. Constraints to this approach include little demand and a lack of resistant material in the marketplace, lack of funds for breeding programs, a broad array of pests, and exceedingly low thresholds that demand high levels of control.^[43] Despite the primordial levels of adoption of resistant ornamental plants, there is reason for optimism because restrictions on the use of pesticides have become more severe and alternatives have been found.^[43] Herms^[43] recently compiled a list of some 50 studies involving more than 20 genera of landscape plants screened for resistance to one or more pest species. In several cases, levels of resistance were high.

Cultural Tactics

Traditional paradigms concerning cultural practices such as fertilization and irrigation are crumbling as a more thorough understanding of the relationships between these practices and pest outbreaks becomes clear. Long-held beliefs, such as that fertilization reduces the susceptibility of plants to pest attack, are being reconsidered. Simplistic notions, such as that environmental stress predisposes landscape plants to pest attack, are being overturned.^[43] Undoubtedly, a more enlightened view of these complex phenomena will help reduce unnecessary inputs of fertilizers and preventative insecticides into landscape ecosystems.

Chemical Controls

During the last decade, the landscape maintenance industry has embraced the use of insecticides categorized as reduced risk. Owing to lower mammalian toxicity, shorter residual activity, and reduced impact on natural enemies, these materials are perceived to be safer for humans, non-target organisms, and the

environment. Horticultural oils have many of these properties and have been readily accepted by growers, landscape managers, and homeowners alike.^[4,5] New classes of systemic chemicals, such as the chloronicotinylns, effectively manage several key insect pests of landscape ornamentals. One chloronicotinyln, imidacloprid, is systemic in plants, active at very low rates, has long residual activity, and is relatively broad-spectrum. Many urban pesticide applicators want to avoid foliar treatments and their associated problems such as drift, odor, public scrutiny, and disruption of nontarget assemblages on vegetation. Systemic insecticides may reduce the exposure of natural enemies on leaves and bark, but omnivorous predators may be killed if they feed on pollen or plant sap. This type of natural enemy disruption has been implicated in outbreaks of spider mites on plants treated with systemics.^[44]

Microbial products and their derivatives such as spinosyns and avermectins offer relative specificity and are minimally disruptive to some groups of natural enemies. Highly specific compounds such as hexythiazox are active against spider mites (Tetranychidae) but not predatory mites (Phytoseiidae). Many of these newer products are compatible with and have been incorporated into IPM programs.

CONCLUSION

Sustainability is the long-term goal in the planning and design of urban landscapes.^[3] Research must focus on ways to restore the natural ecological function of managed landscapes to the maximum extent possible. To accomplish this, the impact of landscape design on communities of plants, pests, and their natural enemies must be elucidated. The effects and interactions of nutrients, water, soils, pollutants, and cultural practices on plant growth and defense must also be defined.^[43] As pesticide regulations and product cancellations restrict the use of and remove compounds from the marketplace, a prediction made a decade ago—that “it will soon become unlawful or impractical to spray vegetation in urban areas with chemicals in common use today”—has largely come true.^[5] This trend has spawned a renewed interest in the use of alternatives to pesticides including resistant germ plasm, cultural practices that enhance plant resistance, refractory landscape design, and the use of biological controls. New and old pesticides must be compatible with biologically intensive pest management if they are to remain in the marketplace. Despite these limitations, the privatization of IPM and PHC is well underway and reductions in pesticide use have been dramatic, ranging from 4.9% to 99.8%.^[5,8,31,45]

REFERENCES

1. USDA-ERS. Floriculture and Environmental Horticulture Situation and Outlook Report—FLO 1997. In *Market and Trade Economics Division*; Economic Research Service, USDA: Washington, DC, 1997; 104 pp.
2. National Gardening Survey; National Gardening Association: South Burlington, VT, 2002; 200 pp.
3. Kuser, J.E. *Handbook of Urban and Community Forestry in the Northeast*; Kluwer Academic Publishing: New York, NY, 2000.
4. Latimer, J.G.; Braman, S.K.; Beverly, R.B.; Thomas, P.A.; Walker, J.T.; Sparks, B.; Oetting, R.D.; Ruter, J.M.; Florkowski, W.; Olson, D.L.; Robecker, C.D.; Garber, M.P.; Lindstrum, O.M.; Hudson, W.G. Reducing the pollution of potential pesticides and fertilizers in the environmental horticulture industry: II. Lawn care and landscape management. *HortTechnology* (Alexandria, VA) **1996**, *6* (3), 222–231.
5. Raupp, M.J.; Koehler, C.S.; Davidson, J.A. Advances in implementing integrated pest management for woody landscape plants. *Annu. Rev. Entomol.* **1992**, *37*, 561–585.
6. Ball, J.; Marsan, P. Establishing monitoring routines and action thresholds for a landscape IPM service. *J. Arboric.* **1991**, *17*, 88–93.
7. Raupp, M.J.; Holmes, J.J.; Sadof, C.; Shrewsbury, P.M.; Davidson, J.A. Effects of cover sprays and residual pesticides on scale insects and natural enemies in urban forests. *J. Arboric.* **2001**, *27* (4), 203–213.
8. Lloyd, J. *Plant Health Care for Woody Ornamentals*; International Society of Arboriculture: Savoy, IL, 1997; 223 pp.
9. Frankie, G.W.; Ehler, L.E. Ecology of insects in urban environments. *Annu. Rev. Entomol.* **1978**, *23*, 367–387.
10. Holmes, J.J.; Davidson, J.A. Integrated pest management for arborists: implementation of a pilot program. *J. Arboric.* **1984**, *10*, 65–70.
11. Raupp, M.J.; Shrewsbury, P.M.; Holmes, J.J.; Davidson, J.A. Plant species diversity and abundance affects the number of arthropod pests in residential landscapes. *J. Arboric.* **2001**, *27* (4), 222–229.
12. Balder, H.B.; Jackel, B.; Pradel, K. Investigations on the populations of beneficial organisms on urban trees. In *International Symposium on Plant Health Care in Urban Horticulture*; Backhaus, G.F., Balder, H., Idczak, E., Eds.; Parey Buchverlag: Berlin, 2000; 47–51.
13. Shrewsbury, P.M.; Raupp, M.J. Evaluation of components of vegetational texture for predicting azalea lace bug, *Stephanitis pyrioides* (Heteroptera: Tingidae), abundance in managed landscapes. *Environ. Entomol.* **2000**, *29* (5), 919–926.
14. Tooker, J.F.; Hanks, L.M. Influence of plant community structure on natural enemies of pine needle scale (Homoptera: Diaspididae) in urban landscapes. *Environ. Entomol.* **2000**, *29* (6), 1305–1311.
15. Owen, D.F. Insect diversity in an english suburban garden. In *Perspectives in Urban Entomology*; Frankie, G.W., Koehler, C.S., Eds.; Academic Press: New York, NY, 1978; 13–29.
16. Taylor, L.R.; French, R.A.; Woiwod, I.P. The rothamsted insect survey and the urbanization of land in great Britain. In *Perspectives in Urban Entomology*; Frankie, G.W., Koehler, C.S., Eds.; Academic Press: New York, NY, 1978; 31–65.
17. Owen, J. Effects of contrived plant diversity and permanent succession on insects in English suburban gardens. In *Urban Entomology: Interdisciplinary Perspectives*; Frankie, G.W., Koehler, C.S., Eds.; Praeger: New York, NY, 1983; 395–422.
18. Hanks, L.M.; Denno, R.F. Natural enemies and plant water relations influence the distribution of an armored scale insect. *Ecology* **1993**, *74* (4), 1081–1091.
19. Leddy (Shrewsbury), P.M. Factors influencing the distribution and abundance of the azalea lace bug. In *Stephanitis pyrioides, in Simple and Complex Landscape Habitats*, Ph.D. Dissertation; University of Maryland: College Park, MD, 1996; 147 pp.
20. Lozzia, G.C. Beneficial insects in integrated pest management on urban trees. Examples in northern Italy. In *Proceedings of the International Symposium on Urban Tree Health*; Lemattre, M., Lemattre, P., Lemaire, F., Eds.; Acta Horticulturae: Paris, 1999; 169–176.
21. Rigamonti, I.E.; Lozzia, G.C. Injurious and beneficial mites on urban trees in northern Italy. In *Proceedings of the International Symposium on Urban Tree Health*; Lemattre, M., Lamattre, P., Lemaire, F., Eds.; Acta Horticulturae: Paris, 1999; 177–182.
22. Raupp, M.J.; Davidson, J.A.; Koehler, C.S.; Hellman, J.L. The concept of key plants in integrated pest management for landscapes. *J. Arboric.* **1985**, *11*, 317–322.
23. Nielson, D.G. Integrated pest management (IPM). *Brooklyn Bot. Gard. Rec.* **1983**, *40* (1), 70–72.
24. Raupp, M.J.; Shrewsbury, P.M. Defining key pests, key plants, and their relationship to vegetational diversity in residential landscapes. In *International Symposium on Plant Health Care in Urban Horticulture*; Backhaus, G.F., Balder, H., Idczak, E., Eds.; Parey Buchverlag: Berlin, 2000; 41–46.
25. Ball, J. Efficient monitoring for an urban IPM program. *J. Arboric.* **1987**, *13*, 174–177.
26. Shetlar, D.J.; Herms, D. *Insect and Mite Control on Woody Ornamentals and Herbaceous Perennials. Bulletin 504*; Ohio State University Extension, 1999; 73 pp.
27. Orton, D.A.; Green, T.L. *Coincide: The Orton System of Pest Management*; Plantsmen's Publication: Flossmoor, IL, 1989.
28. Herms, D.A. Biological clocks. *Am. Nurseryman.* **1990**, *172*, 56–63.
29. Mussey, G.J.; Potter, D.A. Phenological correlations between flowering plants and activity of urban landscape pests in Kentucky. *J. Econ. Entomol.* **1997**, *90* (6), 1615–1627.
30. Brewster, C.C.; Allen, J.C.; Kopp, D.D. IPM from space: using satellite imagery to construct regional crop maps for studying crop–insect interactions. *Am. Entomol.* **1999**, *45*, 105–117.
31. Braman, S.K.; Latimer, J.G.; Robecker, C.D. Factors influencing pesticide use in integrated pest management

- in urban landscapes: a case study. HortTechnology (Alexandria, VA) **1998**, 8 (2), 145–149.
32. Olkowski, W. A model ecosystem management program. Proc. Tall Timbers Conf. Ecol. Anim. Cont. Habit. Manage. **1974**, 5, 103–117.
 33. Sadof, C.S.; Raupp, M.J. Aesthetic thresholds and their development. In *Economic Thresholds for Integrated Pest Management*; Higley, L.G., Pedigo, L.P., Eds.; University of Nebraska Press: Lincoln, 1997; 203–226.
 34. Paine, T.D.; Miller, J.G.; Bellows, T.S., Jr.; Hanks, L.M. Enlisting an underappreciated clientele: public participation in distribution and evaluation of natural enemies in urban landscapes. Am. Entomol. **1997**, 43, 163–172.
 35. U.S. forest service. Gypsy Moth News **1995**, 39, 2–6.
 36. U.S. forest service. Gypsy Moth News **1996**, 42, 1–2.
 37. Ehler, L.E. Biological control of obscure scale (Homoptera: Diaspididae) in California: an experimental approach. Environ. Entomol. **1995**, 84 (4), 95–127.
 38. Landis, D.A.; Wratten, S.D.; Gurr, G.M. Habitat management to conserve natural enemies of arthropod pests in agriculture. Annu. Rev. Entomol. **2000**, 45, 175–201.
 39. Shrewsbury, P.M.; Raupp, M.J. Biological control in specific crops: full IPM programs. In *Biological Control of Arthropod Pests in Protected Culture*; Heinz, K.M., Van Driesche, R., Parrella, M.P., Eds.; Ball Publishing: Batavia, IL, *in press*.
 40. Raupp, M.J.; Hardin, M.R.; Braxton, S.M.; Bull, B.B. Augmentative releases for aphid control on landscape plants. J. Arboric. **1994**, 20 (5), 241–249.
 41. Smith-Fiola, D.C.; Gill, S.A.; Way, R.G. Evaluation of entomopathogenic nematodes as biological control against the banded ash clearwing borer. J. Environ. Hortic. **1996**, 14 (2), 67–71.
 42. Owen, N.P.; Raupp, M.J.; Sadof, C.S.; Bull, B.B. Influence of entomophagous nematodes and irrigation on black vine weevil in *Euonymus fortunei* (Turcz.) Hard. Mazz. beds. J. Environ. Hortic. **1991**, 9, 109–112.
 43. Herms, D.A. Strategies for deployment of insect resistant ornamental plants: substantial hurdles, vast potential. In *Mechanisms and Deployment of Resistance in Trees to Insects*; Wagner, M.R., Clancy, C., Paine, T., Lieutier, F., Eds.; Kluwer Academic Publishing: Dordrecht, Netherlands, 2002; 217–237.
 44. Smith, S.F.; Krishcik, V.A. Effects of systemic imidacloprid on *Coleomegilla maculata* (Coleoptera: Coccinellidae). Environ. Entomol. **1999**, 28 (6), 1189–1195.
 45. Hubbell, B.J.; Florkowski, W.J.; Oetting, R.; Braman, S.K. Pest management in the landscape/lawn maintenance industry: a factor analysis. J. Prod. Agric. **1997**, 10 (2), 223–236.

Lawn-Care Treatments: Weeds

Harlene Hatterman-Valenti

Plant Sciences Department, North Dakota State University,
Fargo, North Dakota, U.S.A.

INTRODUCTION

Weeds are a multibillion dollar problem in golf courses, business and residential lawns, parks, and sports fields.^[1] Weeds aggressively compete for light, water, and nutrients that are essential for turfgrass growth and development. Weeds also detract from the uniformity and playability of turf by producing morphological characteristics that contrast with turf. Annual weeds that invade turf and die upon life cycle completion leave unsightly brown areas. Even turfgrasses can become weeds from leaf blade morphology and growth characteristic differences, e.g., tall fescue (*Festuca arundinacea*) in a Kentucky bluegrass (*Poa pratensis*) turf.

INTEGRATED WEED MANAGEMENT

Turfgrass weed management has evolved in response to various functional, ornamental, and recreational uses for turfgrass. Many grass and broadleaf weeds have evolved and adapted to turfgrass management practices, thereby enabling them to compete and coexist with turfgrass. The most common adaptation of turf weeds is the ability to tolerate continuous defoliation from routine mowing. Grass weeds, similar to turfgrasses, tolerate mowing because their subapical meristems are well below the cutting area of leaf blades. Many broadleaf weeds that persist in turf either have prostrate growth, e.g., prostrate spurge (*Chamaesyce humistrata*), or rosette growth, e.g., dandelion (*Taraxicum officinale*), which keeps the growing point below the mowing height.

Seed germination adaptations have allowed weeds to be opportunists, waiting for favorable germination conditions. Seed dormancy limits germination under favorable conditions and keeps some weed seed viable for several decades, especially under adverse conditions.^[2]

Weed management strategies in turf depend on several factors including climatic region, time of year, age and species of turfgrass, weed density, and specific weed species present. An integrated weed management approach utilizes preventative measures as well as cultural and chemical control methods.

PREVENTION

Weed presence generally indicates a weakened and stressed turf or inadequate perennial weed control prior to turfgrass establishment. The opportunity for weed invasion may have resulted from poor soil physical properties, adverse soil chemical properties, unfavorable environmental conditions, or improper turfgrass maintenance.

Prostrate knotweed (*Polygonum aviculare*) in turf along a sidewalk is generally considered an indication of soil compaction. Salt, used as a deicing agent during winter, also provides a competitive advantage for prostrate knotweed since its germination and growth are favored by high salt concentrations.^[3]

Green kyllinga (*Kyllinga brevifolia*), a perennial sedge, is an increasing problem in the southern United States due to its competitive advantage over bermudagrass (*Cynodon dactylon*) at low mowing heights of 2.5 cm or less.^[4] Green kyllinga requires light for germination; therefore, a dense uniform turfgrass with minimum light penetration to the soil would minimize germination.^[5]

Weed invasions in turf should be managed by controlling perennial weeds prior to turfgrass establishment. Select a turfgrass species/cultivar, blend, or mixture that is well adapted to the specific location. Once established, maintain timely irrigation practices, proper fertilization, correct mowing, and thatch removal plus aeration when needed to ensure a competitive turf. Please refer to Area Extension publications for specific turfgrass management recommendations.

CULTURAL TREATMENTS

Many of the preventative measures previously mentioned are considered cultural control strategies since practices that promote a healthy, vigorous turf will discourage many weeds. Basic cultural practices include mowing, fertilization, and irrigation.

Mowing helps eliminate weeds with upright growth. However, mowing turf below recommended heights will reduce root growth and further stress the grass, thereby opening the canopy for increased weed seed germination (Table 1). Fertilizing dormant turf when

Table 1 Recommended turfgrass mowing heights

| Cutting height (cm) | Common name | Scientific name |
|---------------------|--------------------|--------------------------------|
| 0.5–1.5 | Creeping bentgrass | <i>Agrostis stolonifera</i> |
| | Velvet bentgrass | <i>Agrostis canina</i> |
| 1.5–2.5 | Annual bluegrass | <i>Poa annua</i> |
| | Bermudagrass | <i>Cynodon dactylon</i> |
| | Colonial bentgrass | <i>Agrostis tennuis</i> |
| | Manilagrass | <i>Zoysia matrella</i> |
| | Rough bluegrass | <i>Poa trivialis</i> |
| | Zoysiagrass | <i>Zoysia japonica</i> |
| 2.5–5.0 | Buffalograss | <i>Buchloe dactyloides</i> |
| | Red fescue | <i>Festuca rubra</i> |
| | Centipedegrass | <i>Eremochloa ophiuroides</i> |
| | Carpetgrass | <i>Axonopus affinis</i> |
| | Kentucky bluegrass | <i>Poa pretensis</i> |
| | Italian ryegrass | <i>Lolium multiflorum</i> |
| | Perennial ryegrass | <i>Lolium perenne</i> |
| | Meadow fescue | <i>Festuca elatior</i> |
| | 3.5–7.5 | Bahiagrass |
| Sheep fescue | | <i>Festuca ovina</i> |
| Tall fescue | | <i>Festuca arundinacea</i> |
| St. Augustinegrass | | <i>Stenotaphrum secundatum</i> |
| Fairway wheatgrass | | <i>Agropyron cristatum</i> |
| 7.5–10 | Canada bluegrass | <i>Poa compressa</i> |
| | Smooth brome | <i>Bromus inermis</i> |
| | Timothy | <i>Phleum pratense</i> |
| | Western wheatgrass | <i>Agropyron smithii</i> |

^aRecommended mowing height may vary with a specific cultivar.

weeds are actively growing stimulates weed growth more than turfgrass. Frequent shallow irrigations encourage weed seed germination and turfgrass rooting near the soil surface, thereby making the turf more vulnerable to environmental stresses. This practice also decreases soil aeration and water infiltration, creating wet and compacted conditions. Core aeration, combined with topdressing, is used to alleviate soil compaction and reduce thatch accumulation, but coring when the grass is stressed or not actively growing opens the turf to weed invasions.

Cultural weed control requires thorough knowledge of the turfgrass species being grown as well as correct identification of weeds and their life cycles. Knowing weed life cycles (winter annual, summer annual, biennial, or perennial) and growth habits (bunch type/single stem or spreading) will help determine control measures. Tall fescue is a bunch-type perennial grass, so its removal from a Kentucky bluegrass turf by digging the clumps is a practical cultural control method when the area is small or the infestation is low. However, the same procedure would be futile for control of quackgrass (*Elymus repens*), a spreading rhizomatous perennial grass.

Establishment of a cool-season turfgrass in the northern states is enhanced by early-fall seeding (after perennial weeds have been eliminated) over spring

seeding, especially when winter annual weeds are not a problem. Fall is a time of active cool-season turfgrass growth, which provides extra time for establishment before warm weather that favors germination of many annual weeds.

Correct weed identification and knowledge of weed growth habits may help diagnose the cause of weed encroachment. Several weeds can be indicators of turf problems.^[6] Table 2 contains a list of weeds that often invade turf and soil conditions that favor their encroachment. Correcting these problems will reduce weed reestablishment from dormant seed following weed control treatments.

CHEMICAL TREATMENTS

A survey of professional lawn-care services showed that lawn care sales reached \$21 billion in 2000 and predicted sales to reach \$26 billion by 2005.^[7] Herbicide applications represented a majority of these sales. Herbicide selection and the rate applied depend on several factors such as the turfgrass species, weeds to be controlled, and time of year. Improper herbicide selection and rate and nonuniform application may result in turf injury or inadequate weed control.

Table 2 Problematic soil conditions and weed species that thrive under these conditions

| Soil condition | Common name | Scientific name |
|-------------------------|----------------------|--|
| Wet, poorly drained | Alligatorweed | <i>Alternanthera philoxeroides</i> |
| | Annual bluegrass | <i>Poa annua</i> |
| | Birdseye pearlwort | <i>Sagina procumbens</i> |
| | Little starwort | <i>Stellaria graminea</i> |
| | Mosses | <i>Rhytidiadelphus squarrosus</i> and other species |
| Dry, drought | Sedges | <i>Cyperus</i> spp. |
| | Birdsfoot trefoil | <i>Lotus corniculatus</i> |
| | Black medic | <i>Medicago lupulina</i> |
| | Goosegrass | <i>Eleusine indica</i> |
| | Prostrate spurge | <i>Euphorbia maculata</i> |
| | Yellow woodsorrel | <i>Oxalis stricta</i> |
| Low nitrogen, infertile | Birdsfoot trefoil | <i>Lotus corniculatus</i> |
| | Black medic | <i>Medicago lupulina</i> |
| | California burclover | <i>Medicago polymorpha</i> |
| | Common speedwell | <i>Veronica officinalis</i> |
| | Mouseear hawkweed | <i>Hieracium pilosella</i> |
| | White clover | <i>Trifolium repens</i> |

Herbicides that do not injure one cool-season or warm-season species may injure another turfgrass species. Fenoxaprop is a postemergence herbicide that controls annual grass weeds in established cool-season turf. However, fenoxaprop severely injures creeping bentgrass (*Agrostis stolonifera*) putting greens and only a few bentgrass cultivars maintained at fairway cutting height are sufficiently tolerant to fenoxaprop.^[8] Similarly, herbicide injury to a specific turfgrass cultivar may vary with location. Quinclorac did not significantly injure spring-seeded “Penncross” creeping bentgrass in Indiana or Iowa, while significant injury occurred in North Carolina.^[9] Therefore, always read and follow label instructions prior to herbicide application.

Preemergence herbicides are generally used to control annual weeds. These herbicides provide little control of emerged plants, except dithiopyr, which controls some small annual grasses. Preemergence herbicides must be applied before weed seed germination and must be moved into the weed seed germination zone following application either by rainfall or through irrigation within 1–2 days of application. Herbicides applied too early in the season may not adequately control annual weeds that germinate late in the season because herbicide degradation has occurred.

Preemergence herbicide persistence, on the other hand, may be a concern in southern states where bermudagrass golf greens are usually overseeded with cool-season grasses. Likewise, fall-applied preemergence herbicides for control of winter annual weeds on golf greens may injure dormant bermudagrass the following spring.

Most preemergence herbicides are not safe on newly seeded turf, except siduron and oxadiazon. Siduron may be applied prior to seeding a cool-season turfgrass, but it has a very short residual, so must be reapplied at 3–4-week intervals. Oxadiazon may be applied prior to or immediately after sprigging of bermudagrass or of zoysiagrass in Hawaii only.

Postemergence herbicides control emerged weeds. Repeat applications may be required because most postemergence herbicides provide little soil residual. Most postemergence herbicides selectively control broadleaf weeds in turf. Often mixtures of two or more postemergence herbicides are used to expand the spectrum of broadleaf weeds controlled from a single application. Most turfgrasses are tolerant to these broadleaf herbicides, but consult the labels for exceptions.

Some postemergence herbicides are available in either ester or amine salt forms. Ester forms (short- and long-chain esters) are generally considered more effective than amine forms for control of stressed or hard-to-kill broadleaf species. However, ester forms are also more volatile and present a potential vapor drift hazard to susceptible plants, especially when spraying during warm weather.

Spray drift of postemergence herbicides may cause unintended injury since these herbicides are highly active on many broadleaf species. Simulated drift of 2,4-D at 1/100 the maximum rate used on wheat caused visible injury to grape and reduced grapevine growth.^[10] Off-target drift of spray droplets can be reduced by increasing droplet size, which is accomplished by reducing spray pressure, increasing nozzle orifice size, using special drift reduction nozzles, or adding a drift retardant that increases spray viscosity.

However, susceptible flowers, vegetables, and bushes may be accidentally contacted by herbicide droplets when plants are adjacent to or within the turf area. For example, growth regulator herbicides applied with a spray-gun system injured tomato plants 90 cm from the spray swath.^[11] Therefore, extreme care cannot be overstated when applying these herbicides.

Few herbicides provide selective postemergence grass control in turf, especially to control perennial grass weeds. Three exceptions are chlorsulfuron for control of tall fescue in Kentucky bluegrass turf and atrazine or simazine for cool-season grass control in warm-season turf. Non-selective herbicides with no soil residual may be used for perennial grass control when a selective herbicide is not available. Non-selective herbicides are generally spot-applied to the infested area or may be broadcast over the entire area during turf renovation to kill all green vegetation.

CONCLUSIONS

An integrated weed management program that includes prevention, cultural control, and chemical control is recommended for highly maintained turf. A program should start before turfgrass establishment. A healthy turf is the cornerstone of any weed management program. Weeds thrive when turfgrass struggles. Determine why the weeds have invaded and correct the problem so that weeds cannot reinfest an area.

REFERENCES

1. Watson, J.R.; Kaerwer, H.E.; Martin, D.P. The turfgrass industry. In *Turfgrass*; Waddington, D.V., Carrow, R.N., Sherman, R.C., Eds.; Agronomy Monograph 32; American Society of Agronomy: Madison, WI, 1992; 29–88.
2. Jensen, K. Dormancy patterns, germination ecology, and seed-bank types of twenty temperate fen grassland species. *Wetlands* **2004**, *24* (1), 152–166.
3. St-Arnaud, M.; Vincent, G. Influence of high salt levels on the germination and growth of five potentially utilizable plants for median turfing in northern climates. *J. Environ. Hortic.* **1988**, *6* (4), 118–121.
4. Lowe, D.B.; Whitwell, T.; McCarty, L.B.; Bridges, W.C. Mowing and nitrogen influence green kyllinga (*Kyllinga brevifolia*) infestation in Tifway bermudagrass (*Cynodon dactylon* X *C. transvaalensis*) turf. *Weed Technol.* **2000**, *14* (3), 471–475.
5. Lowe, D.B.; Whitwell, T.; Bridges, W.C. *Kyllinga brevifolia*, *K. squamulata*, and *K. pumila* seed germination as influenced by temperature, light, and nitrate. *Weed Sci.* **1999**, *47* (6), 657–661.
6. Emmons, R.D. Weeds. In *Turfgrass Science and Management*, 2nd Ed.; Delmar Publishing: New York, 1995; 267–286.
7. <http://www.plcaa.org/consumer/industry.asp> (accessed August 2003).
8. Johnson, B.J.; Carrow, R.N. Bermudagrass (*Cynodon* spp.) suppression in creeping bentgrass (*Agrostis stolonifera*) with herbicide—flurprimidol treatments. *Weed Sci.* **1993**, *41* (1), 120–126.
9. Reicher, Z.; Hardebeck, G.A.; Yelverton, F.F.; Christians, N.E.; Bingaman, B.; Turner, J. Tolerance to quinclorac by seedling creeping bentgrass. *Hortscience* **2002**, *37* (1), 210–213.
10. Bhatti, M.A.; Kassim, A.K.; Parker, R.; Al-Khatib, K. Wine grape (*Vitis vinifera*) response to repeated exposure of selected sulfonylurea herbicides and 2,4-D. *Weed Technol.* **1996**, *10* (4), 951–956.
11. Hatterman-Valenti, H.; Owen, M.D.K.; Christians, N.E. Comparison of spray drift during postemergence herbicide application to turfgrass. *Weed Technol.* **1995**, *9* (2), 321–325.

Legal Aspects of Pesticide Applications

Maristella Rubbiani

*Laboratorio di Tossicologia Applicata, Istituto Superiore de Sànita,
Rome, Italy*

INTRODUCTION

In this article, the different responsibilities of pesticide applicators are described. Some of the definitions regarding the qualifying characteristics and requirements of a pesticide applicator are reported, such as the necessity of having the legal requirement (certification), which is needed by the pesticide applicator for the use of a restricted-use pesticide (RUP). A restricted-use indication is applied to pesticides that may cause adverse effects to humans and the environment, and RUP applicators are required to keep records of their use for each application. The purpose of the legal certification is to protect the public and the environmental health, and the actual mechanism for releasing the certification is left to the various state lead agencies working under the Environmental Protection Agency (EPA) rules. Pesticide applicators are required also to follow the instructions provided on the label. The label is approved by the EPA during the evaluation process of the pesticide for registration under the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA). The mandatory indication reported on the label is also described.

DEFINITION OF APPLICATOR

Professional applicators are those who apply or direct the application of pesticides as part of their jobs: lawn care operators, golf course superintendents, indoor pest control operators, and institutional grounds managers working on sites such as parks, schools, resorts, office complexes, right of ways, or industrial locations. Professional applicators are those who apply pesticides to properties other than their own.^[1]

The U.S. Environmental Protection Agency classifies pesticides into two categories, general-use pesticides and restricted-use pesticides, which make up a quarter of total pesticides used and may be applied only by, or under the direct supervision of, trained and certified applicators.^[2]

Certification complements product registration, which designates pesticide products for either general use or restricted use.

A restricted-use classification is applied to products that, when used in accordance with label directions, may cause adverse effects on humans or the environment.

By federal law, anyone who applies restricted-use pesticides must be certified as a competent applicator, or directly supervised by a certified applicator.

CERTIFICATION

Pesticide applicator certification is a legal requirement for persons using restricted-use pesticides in any situation, as well as for those applying general-use products in commercial situations.

The purpose of certification is to protect public health and public welfare and to maintain environmental quality. Certification is a means of ensuring that persons who apply restricted-use pesticides, or make commercial applications, possess the knowledge to do this in a safe and effective manner, avoiding any misuse that could pose a threat to human health and environmental quality.

A certified applicator is an individual who has demonstrated a certain level of competency in the area of pesticide use and application, and is deemed capable of managing the use of pesticide products so as to minimize associated risk.

The actual mechanism of applicator certification is left to the various state lead agencies working under guidelines established by the EPA. The concept of applicator certification received significant support from the EPA through legal procedures for administrative review.

In the event that the EPA determines that the use of a pesticide might pose an undue risk to humans or the environment, a restricted-use classification generally is considered before implementing the more drastic options of cancellation or suspension. This, in a very real sense, serves to emphasize the importance of applicator certification. Trained, knowledgeable, and experienced applicators are regarded as professionals capable of utilizing RUPs in a responsible manner.^[3]

The EPA has set minimum standards for the certification of pesticide applicators. It is the EPA's responsibility to see that minimum standards are met. A lead agency, which is responsible to the EPA for certification training and enforcement, is designated in each state. To become certified, professional applicators must demonstrate, through testing, a practical knowledge of pests related to the category of certification for which one is applying.^[1]

Table 1 Standard for certification of private applicators and commercial applicators

| |
|---|
| Private applicators must show a practical knowledge of: |
| Pest problems and control practices associated with agricultural operations |
| Proper storage, use, handling, and disposal of pesticides and containers |
| Legal responsibility |
| Recognition of common pests and damage caused by them |
| Reading and understanding label and labeling |
| Applying pesticides according to label instructions and warnings |
| Recognizing local environmental situations to be considered during application to avoid contamination |
| Recognizing poisoning symptoms and procedures to follow in case of a pesticide accident |
| Commercial applicators must demonstrate a practical knowledge (determined by a written exam) of: |
| Label and labeling comprehension |
| Safety |
| Environment |
| Pests |
| Pesticides |
| Equipment |
| Application technique |
| Laws and regulations |

Certified applicators are classified as either private or commercial, and there are separate standards for each. A private applicator uses or supervises the use of restricted-use pesticides for the purpose of producing an agricultural commodity. A commercial applicator must demonstrate a practical knowledge of the principle and the practices of pest control and the safe use of pesticides; this competence must be determined by a written exam and, as appropriate, performance testings in different areas (see Table 1). The pesticide applicator certification and training program provides pesticide applicators with the knowledge and the ability to use pesticides safely and effectively. Understanding pesticide product labels and the proper methods of pesticide application is essential in applying pesticides safely and in reducing risks to human health and the environment. Pesticide applicators are trained by state Cooperative Extension Service Pesticide Applicator Training Programs and are certified by pesticide state lead agencies.^[4]

LABEL

Pesticide applicators are required to follow the directions on the label. The EPA approves pesticide labels

in the process of registering a pesticide under the FIFRA. The primary focus in the label approval process for agricultural pesticides involves assessing and regulating the potential risks to humans and the environment posed by such pesticides. The label regulations address the direction for use of a pesticide for the purpose of insuring that pesticide applicators and farmworkers are adequately protected. Additionally, direction for use establishes legal limits as to the amount of pesticide that may be applied and thus allows the EPA to control and to estimate dietary exposure^[5] (see Table 2).

A civil administrative complaint proposing civil penalties is applied in case of misbranding violations such as:

- Violations presenting actual or potential risk of harm to human health or the environment.
- Violations that impede the EPA's ability to fulfill FIFRA goals, or harms the regulatory program.
- Violations resulting from ordinary negligence, inadvertence, or mistake.^[6]

RECORDKEEPING

The U.S. Department of Agriculture's (USDA) Agricultural Marketing Services administers the Federal Pesticide Recordkeeping Program, which requires all certified private pesticide applicators to keep records of their use of federally restricted-use pesticides for a period of 2 years. The pesticide recordkeeping regulations require the certified private pesticide applicator to record the following for each application, within 14 days of the application:

- Brand or product name (trademark name).
- EPA registration number.

Table 2 Definitions on the label^[7]

| |
|---|
| Ingredient statement |
| Chemical name of the active ingredient and trade name |
| Net contents |
| Establishment number |
| EPA registration number |
| Signal word and symbol |
| Danger definitions |
| Emergency first aid measures |
| Information on how to avoid damage to the environment |
| Physical and chemical hazards |
| Direction for use |
| Application rate and quantities to harvest |
| Misuse statement |
| Storage and disposal directions |
| Postharvest interval |

- Total quantity of pesticide applied in common units of measure.
- Date of the application.
- Location of the restricted-use pesticide application.
- Crop commodity, stored product, and site being treated.
- Size of area treated, in units of measures.
- Name of the certified applicator performing and/or supervising the application.
- Certification number of the private applicator.^[8]

CONCLUSION

Attending licensed health care professionals or those acting under their direction, USDA representatives, and State regulatory representatives with credentials have legal access to the records.

No standard federal form is required, so that pesticide recordkeeping can be integrated into the applicator's current recordkeeping schemes.

All certified commercial pesticide applicators will continue to maintain the records they currently

keep under State, Tribal, or Federal regulations. The federal pesticide recordkeeping regulations require all commercial applicators, both agricultural and non-agricultural, to furnish a copy of the data elements required by these regulations to the customer within 30 days of the RUP application.

REFERENCES

1. <http://www.grounds-mag.com> (accessed November 2000).
2. <http://www.epa.gov/oppfead1> (accessed November 2000).
3. <http://www.agcom.purdue.edu/> (accessed November 2000).
4. <http://www.epa.gov/oppfead1/safety/applicators> (accessed November 2000).
5. <http://www.epa.gov/oppmsd1> (accessed November 2000).
6. FIFRA WPS Penalty Policy (interim report final, September 1997).
7. <http://www.cdc.gov/niosh> (accessed November 2000).
8. <http://www.ams.usda.gov/science> (accessed November 2000).

Less Hazardous Alternatives: Promotions

Luis Brenes

Escuela de Agronomía, Universidad de Costa Rica, San José, Costa Rica

Luis Felipe Arauz Cavallini

Escuela de Fitotecnia, Universidad de Costa Rica, San José, Costa Rica

INTRODUCTION

Pest control in the last 50 years has relied heavily on a suppressive approach based on chemical synthetic pesticides. The acute impact on human health has been discussed deeply and some long-term effects might still be unknown (see also in this encyclopedia *Cancers from Pesticides* by Dich and *Chronic Human Pesticide Poisonings* by Kolmodin-Hedman). In addition, disruption of environmental balance is of great concern because pesticides not only affect natural ecosystems but also reduce the stability of agroecosystems, which can cause greater pest problems. Classical examples of this imbalance are the destruction of natural enemies, the development of resistance to pesticides, and the outbreak of new pests that were not previously major problems, such as leaf miners and whiteflies.^[1] Therefore, the use of less hazardous alternatives for pest control is an urgent need from the point of view of human health and environment, but also from an agricultural point of view.

These less hazardous alternatives must also meet economic, social, and technical criteria that can guarantee that they are sustainable and will be adopted by farmers. Effective less hazardous alternatives must not only be developed but also adopted widely. Promotion is the process by which this adoption will take place. Real cases where the adoption of such practices has been successful provide additional lessons.^[2]

URGENT NEED TO DEVELOP ALTERNATIVES

Despite great efforts in the last decades to develop better alternatives, scheduled spraying of synthetic pesticides is still the most common practice for pest control in many parts of the world. The agrochemical industry has made an effort to produce less hazardous pesticides in response to increasingly stringent government regulations, especially in developed countries. Although this can be of benefit to the environment, it is important to point out that despite this effort, some of the newer pesticides are highly toxic to pests, humans, and the environment.

Pesticide control is more common in developed countries where more than 75% of the world's pesticides are used; however, in developing countries where about 4 billion people live, less than 25% of the world's pesticides are applied. Thus developing countries are employing alternative pest controls quite widely. But more countries still consider less hazardous alternatives (biological, cultural, and genetic) as mere supplements to chemical control in many agroecosystems, basically because they have not been developed or tested fully. A preventive approach based on the management of pest habitat and life cycle helps reduce the need for hazardous sprayings, but required information is still lacking for many crop-pest relationships. This is especially true in the tropics because of insufficient funding for research, in addition to the complexity of tropical agroecosystems. High biodiversity makes the plant-pest-predator relationships very complex and difficult to study. Despite these limitations, progress has been made in habitat management for pest control, both in temperate^[3] and tropical^[4] areas.

The participation of the private sector is desirable, but much private investment has focused on products and services that guarantee an economic return to the investor. Seed companies have made a contribution to insect and disease control through resistant varieties, usually based on a few resistance genes. The availability of biological pesticides, such as commercial formulations of *Bacillus thuringiensis*, has greatly increased in the last decade. Hardware and software for weather monitoring linked to pest models are also available commercially. New machines for physical pest control are being developed (e.g., tractor-mounted vacuums can be used to discourage some insects in strawberry production).^[5] Nevertheless, some of the best alternatives for replacing chemical methods are not necessarily profitable for a company to develop (e.g., augmentative biological control, cultural control practices, and durable resistance). Therefore, the involvement of public funds, especially through universities and experiment stations, is necessary but also raises an ethical issue because public funds must be used to develop alternatives that are less attractive to the private sector.

EXTENSION: FARMERS' PERSPECTIVE

Supplying Farmers' Needs

The farmer–agroecosystem–consumer relationship forms the backbone of the food production system. The farmer is the actor who influences the system to obtain the goals earlier defined. When considering new alternatives, the researcher must take into account the farmer's needs, goals, and perceptions. If not, the implementation of this alternative might not be fully achieved. It has been shown that it is better to develop and validate these alternatives together with the farmer, and not only for the farmer.^[6–8]

Decision Making: Goals, Risks, and Perceptions

During recent decades, researchers and extension agents have considered solely the increase in productivity per area as the farmers' main goal. But this is not necessarily true. Farmers, especially small farmers, also have other goals such as the preservation of their lifestyle, financial security, tradition, etc.^[6–8] It must be recognized that the goals of large food industry corporations might not only differ from those of small and medium farmers, but also oppose their interests.

Pest control is used to reduce the risk of a loss in yield or quality due to pests. The way in which a farmer controls a pest will greatly depend on available resources, perceptions, and attitude, toward risk. Risk-averse farmers will prefer a scheduled spraying and thus could be less open to other alternatives.^[9]

Education: The New Generation

Less hazardous alternatives will be more successfully implemented if they are considered not as individual techniques but rather within the conceptual background of a holistic management of the agroecosystem in which prevention is the best approach for pest control. The role of universities in developing a new generation of agricultural professionals committed to this concept is essential. Future problem solvers must not only know basic concepts of ecosystems theory but must also have basic skills on the areas of economics and social sciences. The ability of new professionals to work in multidisciplinary groups will be essential if feasible and sustainable alternatives are to be developed and implemented.

Most modern agricultural curricula in major universities incorporate agroecosystem concepts in their courses. A step further is to include activities common

to several courses into the student's field practice. For example, at the University of Costa Rica, undergraduate courses in entomology, soil science, plant pathology, agroecology, and weed biology share a common field plot, a common case study, and a common field trip, all of which help to build a holistic perspective in agronomy students as early as the second year of their college-level education.

ROLE OF REGULATORY AGENCIES

Legal Instruments to Enforce Less Hazardous Alternatives

Hazardous techniques for pest control are usually less expensive than safer alternatives. In many cases, governments have been responsible because they have subsidized the use of pesticides, thus turning the situation into one of the major constraints to reducing hazardous pest control in the tropics.^[10] However, the environmental and social costs are not included in the economy of the production unit (see *Environmental and Economic Costs of Pesticide Use* by Pimentel and Hart). If governments look for legal instruments to introduce these externalities into the cost of production (e.g., via taxes), hazardous alternatives will be discouraged.

The trend toward fewer but larger farms promotes standardization, which means that production is not necessarily adapted to the particular characteristics of a specific site but standardized to a company's production management plan. Large companies have the challenge to overcome this situation. Modern enabling technologies, such as precision agriculture, are working their way into commercial farming. Governments have the challenge to protect small farmers and promote their empowerment.

ROLE OF THE MARKET

The consumer's right to choose can exert a great influence on the production system. If consumer's choice is guided by cosmetic standards, unnecessary pest control will occur at the farm level. If the consumer is informed and understands personal responsibility, one can choose products that come from more sustainable systems that used less pesticide or alternative non-chemical controls.

In the last three decades, the consumer's right to choose has found a practical instrument in environmental and social certifications (labels). These labels have become market-driven forces that stimulate the adoption of less hazardous alternatives through

verified systems such as the Organic Agriculture, ISO-14001, Fair Trade, and others. For other references, please see the following entries in this encyclopedia:

- *Non-chemical or Pesticide Free Farming* by Rundgren and Källander.
- *Organic Agriculture* by Delate.
- *Organic Farming* by Frick.
- *Pest Management in Ecological Farming* by Dinham.
- *Pest Management in Organic Farming* by Gallo.

CONCLUSION

Promotion of less hazardous alternatives requires efforts in the different components of the agricultural sector. The academia has the responsibility to help generate, together with the farmers, the knowledge needed to fill the gaps between agroecological theory and practical pest control, and to develop working technologies for sustainable crop protection. In addition, it has a duty to improve the agroecological education of the new generation of agricultural professionals. Governments should promote less hazardous activities through regulation and extension. The industry can bring about the needed commercial technologies. But it is the educated consumer preference that will ultimately decide the commercial feasibility of safe alternatives to pest control, and therefore their adoption by the farmers.

REFERENCES

1. Pimentel, D.; Greiner, A. Environmental and socio-economic costs of pesticide use. In *Techniques for Reducing Pesticide Use*; Pimentel, D., Ed.; John Wiley and Sons, Ltd.: UK, 1997; 51–78.
2. Thrupp, L.A. New partnerships for sustainable integrated pest management: key social and institutional factors to implement agroecological alternatives. In *Book of Abstracts*, International Conference on Pesticide Use in Developing Countries: Impact on Health and Environment, Costa Rica, Feb 23–28, 1998; Pesticide Program: Development, Health and Environment, Universidad Nacional: Costa Rica, 1998.
3. Landis, D.A.; Menalled, F.D.; Lee, J.C.; Carmona, D.M.; Pérez-Valdés, A. Habitat Management to Enhance Biological Control in IPM. In *Emerging Technologies for Integrated Pest Management*; Kennedy, G.G., Sutton, T.B., Eds.; APS Press: St. Paul, MN, 2000; 226–239.
4. Mezzón, R.G.; Chinchilla, C.M. Plant species attractive to beneficial entomofauna in oil palm (*Elaeis guineensis* Jacq.) plantations in Costa Rica. *ASD Oil Palm Pap.* **1999**, *19*, 1–39.
5. Plucknett, D.L.; Winkelmann, D.L. Technology for sustainable agriculture. *Sci. Am.* **1995**, *273* (3), 148–152.
6. World Neighbors. In *Two Ears of Corn: A Guide to People-Centered Agricultural Development*; World Neighbors: Oklahoma, USA, 1985.
7. Chambers, R., Pacey, A., Thrupp, L.A., Eds.; *Farmer First: Farmer Innovation and Agricultural Research*; Intermediate Technology Publications: UK, 1991.
8. Nelson, R.; Orrego, R.; Ortiz, O.; Tenorio, J.; Mundt, C.; Fredrix, M.; Vien, N.V. Working with resource-poor farmers to manage plant diseases. *Plant Dis.* **2001**, *85* (7), 684–695.
9. Palti. Farmers' perceptions of pest and disease control. In *Advisory Work in Crop Pest and Disease Management*; Palti, J., Ausher, R., Eds.; Springer-Verlag: Berlin, 1986; 19–20.
10. Botrell, D.G. Applications and problems of integrated pest management in the tropics. *J. Plant Prot. Trop.* **1987**, *4* (1), 1–8.

Lettuce Diseases: Ecology and Control

Krishna V. Subbarao

Department of Plant Pathology, (U.S. Agricultural Research Station), University of California at Davis, Salinas, California, U.S.A.

Steven T. Koike

UC Cooperative Extension, Salinas, California, U.S.A.

INTRODUCTION

The transformation of lettuce from a wild weed into a staple salad vegetable is in itself a fascinating story. Cultivated lettuce (*Lactuca sativa*) is thought to have originated around the Mediterranean from *Lactuca serriola*, the prickly lettuce. The earliest written records of Herodotus indicate lettuce cultivation dating back to 550 B.C., though its appearance in stylized paintings in 4500-year-old Egyptian tombs indicates an even more ancient history. Lettuce was brought into the New World by Christopher Columbus.^[1] Early in the settlement of North America by Europeans, lettuce was grown in market gardens near cities and in home gardens. The development of the western shipping industry during the early 20th century transformed lettuce into an economically viable vegetable in the United States.^[1]

TYPES OF LETTUCE

Since its domestication as a vegetable crop, a medley of lettuce types have been developed. The cultivation of specific types of lettuce is dictated by a combination of geography, climate, consumer preference, and market forces. The most common types of lettuce grown throughout the world are crisphead, romaine, green or red leaf, butterhead, Batavia, Latin, stem, and oilseed. Crisphead lettuce is also referred to as “iceberg,” after the name of a cultivar grown extensively in the mid-20th century. The four principal types of lettuce that predominate commercial production in the United States are crisphead, romaine, green or red leaf, and butterhead. The characteristics that distinguish these four types are the formation of a head, its shape and size, and texture.^[1]

Lettuce is rich in vitamins A and C, and minerals such as calcium, potassium, and sodium. Crisphead lettuce has by far the least amount of vitamins and minerals followed by increasingly higher amounts in the butterhead, leaf, and romaine types.^[1] The generalized growth stages of a crisphead lettuce are depicted in Fig. 1.

PRODUCTION OF LETTUCE

Major lettuce-producing countries of the world include Belgium, France, Germany, Great Britain, Italy, the Netherlands, Spain, and the United States. Lettuce production also occurs to a lesser extent in Australia, Japan, Israel, and Taiwan.^[2] The United States is the biggest producer of lettuce, with an aggregate production area of 108,000 ha and a farmgate value of more than \$1 billion annually. Crisphead lettuce accounts for 48% of the production area and the leaf, butterhead, and romaine types for the balance of production. The increasing popularity of ready-to-eat salad mixes in recent years has warranted increased production of the latter types of lettuce.

In the United States major production of all lettuce types is concentrated in California and Arizona. California produces more lettuce than any other country in the world, and Arizona produces more than most other countries. Annually, lettuce production in these two states alone accounts for nearly 90% of the total U.S. production, with California contributing approximately 75% of this total. The remaining 10% of U.S. lettuce is grown in Colorado, Florida, Michigan, New Mexico, New York, New Jersey, and Ohio.^[1]

In California, major lettuce production is concentrated in the coastal valleys of Salinas and Santa Maria. Although lettuce production can occur year-round in both these valleys, in Salinas, it is interrupted by the “lettuce-free” period between December 7 and December 21. This mandatory period is imposed to prevent lettuce mosaic in succeeding lettuce crops. Significant production occurs during late fall and winter in the San Joaquin, Imperial, and Palo Verde valleys in California. Most of the Arizona production occurs during the late fall and winter^[1] and is concentrated in the western part of the state.

LETTUCE DISEASES

Diseases are a significant limiting factor for lettuce production in many parts of the world when resistant

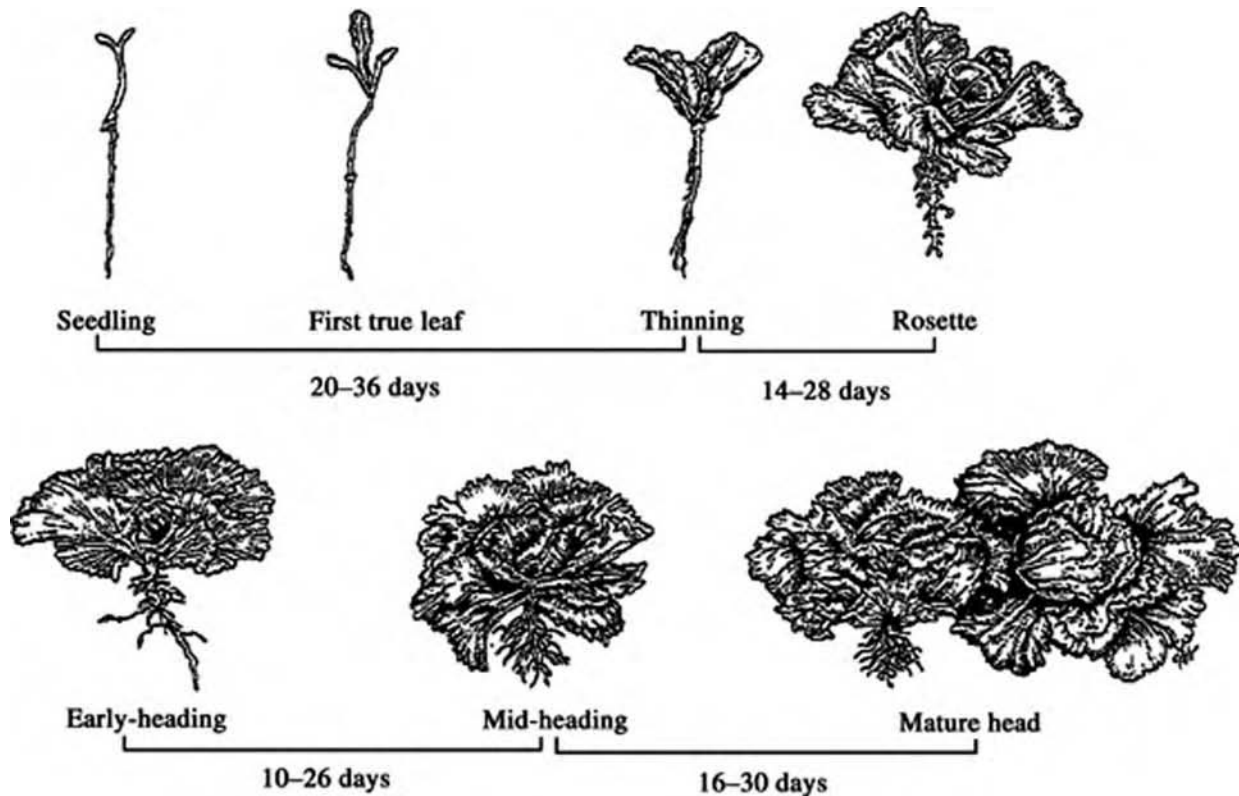


Fig. 1 Growth stages of crisphead lettuce. (From Ref.^[1].)

cultivars are unavailable or not planted. The nature and frequency of these diseases depends on the local conditions. There are nearly 75 known lettuce diseases of diverse etiologies.^[3] Lettuce diseases, as with any other plant disease, are the result of interactions among the lettuce plant, the pathogen (bacterium, fungus, virus, mycoplasma, nematode, adverse abiotic factors, and environmental conditions that either predispose the plant or favor the pathogen), vector, and environmental conditions that favor disease development. Abiotic conditions, such as saline soil, nutrient deficiencies, waterlogged soil, etc., are severe enough to cause diseases in the absence of a pathogen. Lettuce requires relatively abundant and constant soil moisture throughout its growth period. Variations in irrigation and cultural practices for the crop, particularly during head formation stages, can have a severe impact on productivity and lettuce quality.

General descriptions of fungi, bacteria, viruses, and nematodes are available elsewhere in the Encyclopedia, and hence only the description of phytoplasmas is presented here. Phytoplasmas, hitherto referred to as mycoplasma-like organisms, cause certain yellow diseases in plants. Phytoplasmas are submicroscopic entities with highly pleomorphic cells ranging from 70 to 1000 nm in diameter. Phytoplasmas are found in the phloem tissue and are transmitted from plant to plant

by grafting, the parasitic plant, dodder, and the feeding activities of certain insects, mostly leafhoppers. Mycoplasma-like organisms can be distinguished from viruses in that they are not mechanically transmissible and are sensitive to antibiotics such as tetracycline.^[1]

Because lettuce is consumed as a fresh salad, the crop is either marketed as whole heads or in salad mixes after limited processing shortly after harvest. The appearance, size, shape, color, and weight of the produce are all important considerations when whole heads are marketed and diseases that alter these characteristics become economically important and their management imperative.^[2] Thus, losses caused by diseases can be both qualitative through aesthetic damages and quantitative through direct yield losses. In either case, the damage threshold for lettuce is very low as it is a fresh vegetable. Examples of such diseases are anthracnose, bacterial leaf spot, lettuce big vein, corky root, downy mildew, lettuce dieback, lettuce mosaic, powdery mildew, varnish spot, etc. (Table 1). Other diseases that either stunt the plants enough to render them nonharvestable or outright kill plants also result in extensive, direct yield losses. Examples of this type of diseases are *Phoma* basal rot, gray mold, lettuce drop, *Fusarium* wilt, *Verticillium* wilt, etc. (Table 1). In contrast, postharvest decays are caused by pathogens initiated in the production fields or by

Table 1 Common diseases of lettuce, causal agents, symptoms and their general management

| Disease | Pathogen | Symptoms | Management |
|---------------------|---|---|---|
| Anthraxnose | <i>Microdochium panattonianum</i> | Tan leaf spots and lesions on leaves Spots are irregular and angular in shape White-pink spores develop in the spot centers Disease is initiated from soil-borne inoculum, but later spreads via airborne spores | Apply fungicides Select fields without a history of the disease |
| Aster yellows | Aster yellows phytoplasma | Severe stunting and yellowing Twisting of the plant axis Orange-pink latex deposits on leaf surface | Avoid planting in areas where the phytoplasma is present in plant hosts and where leafhoppers occur |
| Bacterial leaf spot | <i>Xanthomonas campestris</i> pv. <i>vitiatis</i> | Water-soaked, angular leaf spots Spots rapidly turn black | Use pathogen-free seed The bacterium can survive in soil, so avoid planting consecutive lettuce crops |
| Bottom rot | <i>Rhizoctonia solani</i> | Spots occur mostly on older foliage Petioles and leaves in contact with soil develop irregular, brown, sunken lesions Lesions later break down and decay, with the rot moving up into the plant Yellow patches on main tap roots | Avoid using sprinkler irrigation Avoid planting in fields having undecomposed crop residues |
| Corky root | <i>R. suberifaciens</i> | Patches later turn brown-green and rough and corky, plants can be stunted | Use resistant cultivars; avoid using excess nitrogen; transplant the lettuce instead of direct seeding |
| Downy mildew | <i>B. lactucae</i> | Yellow, irregularly shaped leaf lesions White mycelium and conidia develop on undersides of leaves Lesions later dry and become brown | Plant lettuce in the spring Practice good crop rotation Use resistant cultivars Apply fungicides |
| Fusarium wilt | <i>Fusarium oxysporum</i> f. sp. <i>lactucum</i> | Aboveground stunting, collapse | Select fields without a history of the disease |
| Gray mold | <i>Botrytis cinerea</i> | Brown discoloration of vascular tissue Crowns in contact with soil develop an orange-brown, very soft decay | Reduce damage to lettuce transplants Keep bed tops dry Do not plant into existing crop residues Apply fungicides |
| Lettuce big vein | Mirafiori lettuce virus | Gray sporulation grows on decayed tissue Plants eventually collapse Distorted, enlarged leaf veins that are abnormally cleared; leaf and head formation can likewise be distorted | Disease is typically more severe in spring, so avoid infested fields until later in the summer Use resistant cultivars |

(Continued)

Table 1 Common diseases of lettuce, causal agents, symptoms and their general management (Continued)

| Disease | Pathogen | Symptoms | Management |
|-------------------|-------------------------------|---|---|
| Lettuce dieback | Lettuce necrotic stunt virus | Lower leaves of romaine cultivars turn bright yellow, sometimes with brown necrotic areas and/or veinal necrosis Newer inner leaves are green, but often have pinpoint chlorotic flecks within minor veins; plants can be severely stunted | The pathogen is an unusual virus that has no known vector; the virus is spread in water and soil; plant remains in fields without a history of the disease |
| Lettuce drop | <i>S. minor</i> | Aboveground stunting, collapse Soft and rotted crowns; profuse white mycelium and small ($\frac{1}{8}$ in.) black sclerotia form on crowns | Apply fungicides at thinning Select fields without a history of the disease Deep plow to bury sclerotia Practice good crop rotation Apply fungicides at rosette stage |
| Lettuce drop | <i>S. sclerotiorum</i> | Aboveground stunting, collapse Soft and rotted crowns; profuse white mycelium and large ($\frac{1}{4}$ in. or larger), black sclerotia form on crowns; airborne spores also cause leaf infections | Deep plow to bury sclerotia Practice good crop rotation Apply fungicides at rosette stage |
| Lettuce mosaic | Lettuce mosaic virus | Plants are stunted and chlorotic Leaf margins are more serrated than normal Edges of leaves curl away from center of plant Some mosaic, mottling, and necrosis can occur | Use pathogen-free seed Remove weed and reservoir hosts Manage the aphid vector Some resistant cultivars are available |
| Phoma basal rot | <i>Phoma exigua</i> | Plants are stunted, chlorotic, and wilt Crown tissue in contact with soil develops cankers Cankers are dark brown to black, sunken, and are not soft in texture | Avoid planting romaine in infested fields Apply fungicides at thinning stage |
| Powdery mildew | <i>Erysiphe cichoracearum</i> | Powdery white mycelial growth develops on leaves Severely infected foliage becomes distorted and eventually dries out | Apply fungicides |
| Varnish spot | <i>Pseudomonas cichorii</i> | Symptoms only develop on varieties that form enclosed heads | Avoid planting lettuce that form enclosed heads If water sources are contaminated, do not use sprinklers |
| Verticillium wilt | <i>Verticillium dahliae</i> | Inner leaves develop firm, shiny brown lesions Lesions are irregular in shape with angular edges Aboveground stunting, collapse Black discoloration of vascular tissue | Water treatments may provide some control Select fields without a history of the disease |

abiotic factors. Such diseases reduce shelf-life or make lettuce less desirable for consumption. Examples of postharvest diseases include gray mold (Table 1), brown stain, pink rib, and russet spotting.

While some lettuce pathogens are unable to survive from season to season, others survive in soil or on plant debris for extended periods of time. In general, few bacterial pathogens survive for prolonged periods (with the exception of corky root pathogen, *Rhizomonas suberifaciens*) but most survive for limited periods as saprophytes on plant debris or roots, or directly in the soil. Unlike bacteria, fungal pathogens produce resilient survival structures on infected lettuce tissues; these structures are released into the soil by tillage operations and through decomposition of the infected material. These survival structures (chlamyospores, sclerotia, microsclerotia, etc.) can withstand high or low temperature, dry or wet conditions, and the absence of suitable hosts. Most viruses survive either in vectors or on alternate hosts, although in one case (lettuce necrotic stunt virus), the virus can survive for extended periods in soil or water.

A number of cultural and environmental factors determine the severity of lettuce diseases.^[3] The first is the availability and density of inoculum. Some lettuce pathogens are seed-borne and using clean seed either eliminates or reduces diseases caused by them [e.g., lettuce mosaic virus (LMV), bacterial leaf spot, Verticillium wilt]. Lettuce crops are mostly grown in monocultures that invariably lead to accumulation of inoculum in the soil and exacerbation of diseases such as corky root, lettuce drop, Verticillium wilt, Fusarium wilt, etc. Second, the type of irrigation can impact on the type and severity of lettuce diseases. In general, furrow and sprinkler irrigations increase the severity of lettuce diseases compared with subsurface or surface drip irrigations (downy mildew, lettuce drop, varnish spot, bacterial leaf spot, Verticillium wilt, etc.).^[4] Third, as the appearance of the lettuce head is less important for salad mixes, increasing plant density offers an ideal opportunity to produce more lettuce per unit area. Thus, the 2 m-wide bed configuration with five to six rows of lettuce and two to three surface drip lines on each bed is taking hold as a standard production practice in recent years. Compared with the standard 1 m-wide bed configuration, irrigation under the 2 m-wide bed configuration is likely to increase soil moisture in the upper soil profiles. The higher plant density on 2 m-wide beds is likely to result in greater moisture retention under the plant canopies. This in turn may increase the incidence of lettuce drop caused by the airborne ascospores of *Sclerotinia sclerotiorum* and the soil-borne sclerotia of *Sclerotinia minor*, and the severity of downy mildew.

The choice of lettuce type and cultivar can have a significant impact on certain lettuce diseases.^[1] For

example, Phoma basal rot and lettuce dieback are more severe on romaine lettuce than on crisphead lettuce. Lettuce cultivars have different resistance genes to the various pathotypes of *Bremia lactucae*.

LETTUCE DISEASE MANAGEMENT

Effective management of lettuce diseases depends on a thorough knowledge of the pathogen, the host plant, the environment, vectors, if any, and their interaction.^[5] The precise identity of the causal agent is of paramount importance in devising management strategies. Disease management options should be based on economical considerations, i.e., the value of the crop saved should exceed the cost of control. Managing highly destructive diseases such as downy mildew can be essential for worthwhile yields, and in such cases routine applications of management options early in the season may be advisable. In addition to being economically sound, management strategies should be simple, safe, and sufficiently effective to reduce diseases to acceptable levels. However, few management options possess all these desirable qualities. To achieve these desirable qualities, an integration of several management strategies is usually required.

Host resistance against many pathogens can be long lasting and environmentally sound. Cultural controls are many and have tremendous potential. The key is to not only develop an effective method, but also to implement it into the current production systems for rapid acceptance. If major changes in cultural practices are required, the practices may be adopted more slowly by the growers. At the same time most cultural practices will not need any regulatory consideration; thus, implementation after development can be extremely fast.^[6] The major limitations of chemical controls are that the degree of control may be unacceptably low in a high-value crop like lettuce, and the need for meeting extensive regulatory requirements. The major limitations of biological controls are the often lower level of control and the higher cost. While each method of control by itself may not provide the desired levels of control, an integration of host resistance, cultural, legislative, chemical, and biological controls is likely to result in successful management of specific diseases.

Successful integration of the different strategies is illustrated by the management of LMV in California. Collaboration between research scientists, growers, and regulatory agencies has resulted in integrated strategies for maintaining LMV at minimal levels. The first line of defense in this integration is to screen all lettuce seed for planting in the Salinas Valley for seed-borne

LMV using enzyme-linked immunosorbent techniques, and planting only those seed lots with less than 1 seed infested per 30,000. Second, weeds that are reservoirs of LMV are thoroughly controlled during lettuce production. Third, infected lettuce plants that are also a source of the virus are plowed under soon after harvest. Fourth, an annual host-free period is enforced for 2 weeks in December to prevent year-to-year buildup of LMV. Since LMV is an obligate pathogen, this step is highly effective. Fifth, fields prone to developing lettuce mosaic owing to proximity to virus reservoirs are discouraged from planting lettuce. Sixth, cultivars resistant to LMV are available and contribute to this integrated program. Finally, spraying for the aphids that vector LMV does not prevent the disease because aphids transmit the virus before the insecticides kill the insects. However, aphid control helps slow down LMV spread and is therefore practiced.^[1]

REFERENCES

1. Davis, R.M.; Subbarao, K.V.; Raid, R.N.; Raid, E.A. *Compendium of Lettuce Diseases*; American Phytopathological Society: St. Paul, MN, 1997.
2. Ryder, E.J. Origin and history of lettuce, types of lettuce, and production. In *Compendium of Lettuce Diseases*; Davis, R.M., Subbarao, K.V., Raid, R.N., Kurtz, E.A., Eds.; American Phytopathological Society: St. Paul, MN, 1997; 1–8.
3. Subbarao, K.V. Progress toward integrated management of lettuce drop. *Plant Dis.* **1998**, *82*, 1068–1078.
4. Xiao, C.L.; Subbarao, K.V.; Schulbach, K.F.; Koike, S.T. Effects of crop rotation and irrigation on *Verticillium dahliae* microsclerotia in soil and wilt in cauliflower. *Phytopathology* **1998**, *88*, 1046–1055.
5. Agrios, G.N. *Plant Pathology*, 5th Ed.; Academic Press: San Diego, CA, 2004.
6. Subbarao, K.V. Methyl bromide alternatives: meeting the deadlines—introduction. *Phytopathology* **2002**, *92*, 1334–1336.

Locust Control by Early Identification of Breeding Sites

Arnold van Huis

Laboratory of Entomology, Wageningen University, Wageningen, The Netherlands

INTRODUCTION

Grasshopper species that change their behavior as a reaction to crowding are called locusts. At low population densities, locusts exist in the solitary phase; and at high densities, they exist in the gregarious phase (Fig. 1). Solitary locusts occur in restricted (recession) areas. When conditions become favorable and the population increases, locusts may become gregarious, and are then capable of migrating further into other (often agricultural) areas, the so-called invasion areas. The preventive control strategy strives to find and destroy gregarious populations in restricted outbreak areas before they invade agricultural lands and destroy crops. Can outbreak areas be delimited either by knowing where gregarious breeding has occurred or by knowing what characterizes an outbreak area capable of generating a plague? This article analyzes the value of early identification of locust outbreaks to effectuate control.

The outbreak concept is different for several locust species, as will be illustrated for the red locust, the tropical migratory locust, the brown locust, the extinct Rocky Mountain locust, and the desert locust. More emphasis will be given to the desert locust, as this is the world's most important locust species.

RED LOCUST

The red locust, *Nomadacris septemfasciata* Serv., has ecologically well-defined outbreak areas – they all are large grass plains subject to annual flooding in areas of closed or highly impeded drainage. Two outbreak areas were implicated in the initiation of the 1930–1944 plague: marshes in Zambia and a valley in Tanzania. Gregarization has occurred in a number of other ecologically similar areas from which small swarms escaped, but plagues did not result. The current control strategy is to prevent swarm escape from the recognized breeding areas, as this is technically and politically easier to achieve than plague termination by massive control.^[1]

AFRICAN MIGRATORY LOCUST

The tropical or African migratory locust, *Locusta migratoria migratorioides* (R&F), in its solitary

form is found over many widely separated areas of Africa. However, large spreading plagues are associated with flood plains, as found in particular in the Middle Niger in Mali, the Lake Chad basin, and Sudan and Madagascar. The flooding of the outbreak area enables locusts to breed not only during the rainy season, but also when the floods recede, allowing for up to five generations a year in Mali; this ensures continued population growth over the year.^[2] During the last century, there were only two plagues in Africa. The last one (1928–1941) invaded most of sub-Saharan Africa and was traced back to the flood plains of the middle Niger in Mali. In the breeding areas of Mali, Lake Chad, Sudan, and Madagascar, systemic burning, grazing, cultivation, and irrigation favor the multiplication of locusts and are probably responsible for maintaining locust numbers during recessions, which can lead to outbreaks during the rainy season.^[3] The development of such solitary populations in these suspect areas should be monitored.

AUSTRALIAN PLAGUE LOCUST

Most outbreaks of the Australian plague locust, *Chortoicetes terminifera* (Walker), occur within a region of some million km² in the Channel Country of southwest Queensland and adjacent areas of South Australia and New South Wales. Large locust populations can develop following rainfall in this region. If undetected, swarms may migrate into the agricultural areas of New South Wales, South Australia, Queensland, and Victoria. Plague populations can develop within one or two years if good rains fall in the interior, allowing them to complete two to four generations per year. As long as populations can complete two to three generations per year, they remain at plague levels. A prolonged dry period will reduce population levels. The strategy of control is to delay plague development by controlling bands and swarms until normal dry conditions intervene and populations decline naturally. Outbreaks require control about every two years.

BROWN LOCUST

The brown locust, *Locustina pardalina* (Walker), occurs during the solitary phase in the desert and

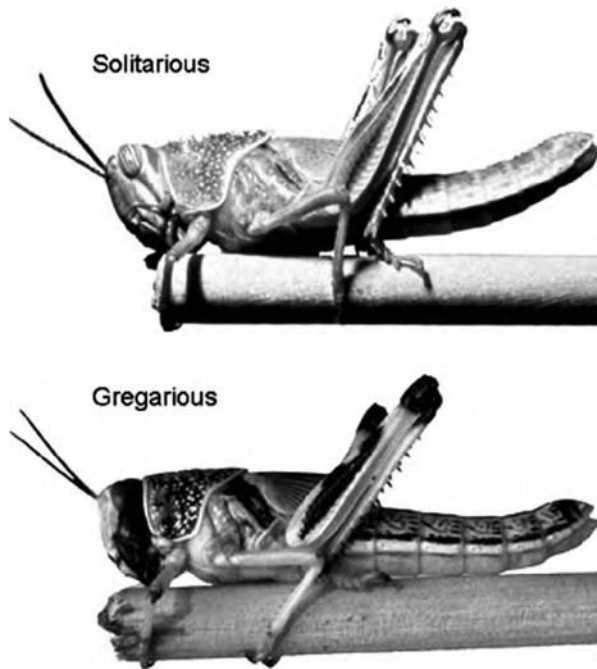


Fig. 1 The solitarious and gregarious phase of the desert locust.

Source: Photograph courtesy Compton Tucker, NASA GSFC.

semi-desert Karoo area of about 250,000 km² of South Africa. Plagues in the wider southern African region (including neighboring Namibia, Botswana, and Zimbabwe) originate from this restricted source area. The outbreak process is most sensitive to rainfall in the early summer period, particularly in December.^[4] The locust is very well-adapted to its semi-arid environment as the eggs become quiescent or enter diapause, remaining viable up to 15 months. Outbreaks have been associated with rainfall after long periods of drought. The shift from solitarious to gregarious locusts occurs from one generation to the next, requiring the application of pesticides over large areas. It is difficult to predict outbreaks because they develop so rapidly.

ROCKY MOUNTAIN LOCUST

The Rocky Mountain locust, *Melanoplus spretus* (Walsh), was the most serious agricultural pest in the western United States and Canada before 1900. In the late 1800s, the species began to decline and became extinct just after 1900.^[5] The invasion area of this locust, believed to have existed in a solitarious and gregarious phase, covered some 5.5 million km². The recession (outbreak) area of this locust was considered only 70,000 km², but within this region, the area was further restricted to the oviposition sites, i.e., riparian

habitats. Anthropogenic changes brought about in these oviposition sites are believed to have played a key role in the extinction of the species.

DESERT LOCUST

Outbreaks of the desert locust, *Schistocerca gregaria* Forskål, may occur anywhere in the whole recession area, estimated at 14.6 million km². This is already about half that liable to be invaded by swarms, with the invasion area estimated at 29.3 million km² (Fig. 2). In this recession area, large scattered populations are potential sources for outbreaks. The outbreak area during recessions can be restricted by: (1) seasonal breeding patterns; (2) the occurrence of rains; (3) flying locusts being concentrated by wind convergence; and (4) patterns of habitat, soil moisture, and vegetation. Gregarization has been known in a number of areas and occurrences of hoppers during recessions have been mapped.^[2,6] The problem is not only that the number of records of locust breeding may reflect human population densities, but also that these suspected areas still cover an immense territory.

These areas of gregarization are not confined to a single ecological unit. For example, the 100,000 km² area of the Tamesna of Niger is a patchwork of distinct geomorphological and ecological units.^[6] The desert provides locusts with a wide range of habitats with great seasonal variability, some of them offering very favorable conditions. However, they are not able to support a stationary locust population because many such ecological islands are ephemeral.

The definition of outbreaks includes the formation of bands and swarms, meaning that gregarizing populations consisting of grouped and scattered individuals do not qualify as outbreaks. A distinction should also be made between localized outbreaks and the more numerous and widespread contemporaneous outbreaks that initiate upsurges. The chief factor determining the commencement of an upsurge is exceptionally high rainfall, which allows for the development of two successive generations in one breeding season. The plague of 1968–1969 has been analyzed thoroughly, with the critical outbreak occurring in 1966–1967 in the Arabian Peninsula, allowing three generations in the minimum possible time.^[7] Prior to the last plague, from 1986 to 1988 there had been 12–13 generations, meaning two generations during winter and spring and two in the summer.^[8] An estimated eight generations occurred before the 1992–1994 upsurge, as well as during the 2003–2005 upsurge. Therefore, it is more appropriate to use the term “outbreak conditions,” rather than “outbreak areas” for this insect. The boundaries between the terms “outbreaks” and “upsurges” are

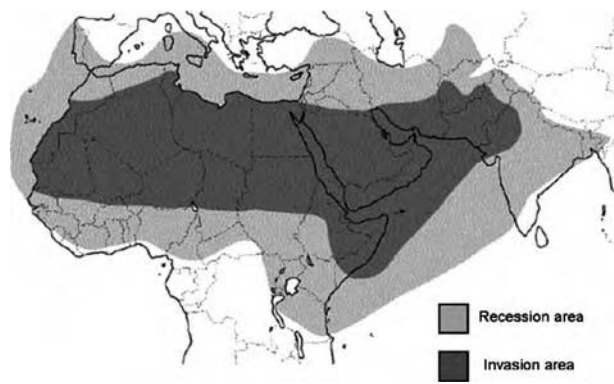


Fig. 2 Recession and invasion area of the desert locust.
 Source: From FAO (see Ref.^[9]).

subjective and imprecise, so that one person's outbreak may be another person's early upsurge.

The efficacy of treating early populations of the desert locust and the optimum stage for interventions are disputed. At the very early stage, only a small part of the population is aggregated in treatable targets. In addition, these small groupings occur over large areas and are difficult to find. To treat them effectively would be unacceptable in terms of costs and environmental pollution. With each subsequent season, the population becomes more gregarious and more locusts exist in a smaller area. Treating early or waiting becomes a balance between control effectiveness in terms of population reduction, what can be achieved and afforded in terms of resources, and how environmental side effects can be minimized.

CONCLUSIONS

The success of early breeding site identification in terms of control depends on whether the breeding sites are restricted in area. Outbreaks of the Australian plague locust and the desert locust do not occur in defined

restricted breeding sites, and may occur anywhere in the recession area when conditions become favorable. Early identification does not automatically ensure successful control. Decisions to control depend on whether targets are treatable, meaning coherent enough and concentrated in bands and swarms. However, in addition to technical conditions, environmental, financial, and political considerations often play a role in decisions regarding whether and when to control. Research on recession populations is difficult, but strongly recommended in order to gain a better understanding of the factors involved in the initiation of upsurges and plagues.

REFERENCES

1. Symmons, P.M. The prevention of plagues of the red locust, *Nomadacris septemfasciata* (Serv.). *Acrida* **1978**, *7*, 55–78.
2. Steedman, A. *Locust Handbook*; Natural Resources Institute: Chatham, 1990; 204 pp.
3. Farrow, R.A. Outbreaks of tropical migratory locust, *Locusta migratoria migratorioides* (R&F). *Insect Sci. Applic.* **1987**, *4–6*, 969–975.
4. Todd, M.C.; Washington, R.; Cheke, R.A.; Kniveton, D. Brown locust outbreaks and climate variability in southern Africa. *J. Appl. Ecol.* **2002**, *39*, 31–42.
5. Lockwood, J.A.; DeBrey, L.D. A solution for the sudden and unexplained extinction of the rocky mountain grasshopper (Orthoptera: Acrididae). *Environ. Entomol.* **1990**, *19*, 1194–1205.
6. Popov, G.B. *Atlas of Desert Locust Breeding Habitats*; Food and Agriculture Organization of the United Nations: Rome, Italy, 1997; 122 pp.
7. Bennett, L.V. Development of a desert locust plague. *Nature* **1975**, *256* (5517), 486–487.
8. Skaf, R.; Popov, G.B.; Roffey, J. The desert locust: an international challenge. *Phil. Trans. R. Soc. Lond. B* **1990**, *328*, 525–538.
9. FAO. *The Desert Locust Guidelines*. Biology and behaviour: Rome, FAO, 1997; Vol. 1.

Lygus Bug Management by Alfalfa Harvest Manipulation

Charles G. Summers
Shannon C. Mueller

Department of Entomology, University of California, Parlier, California, U.S.A.

Peter B. Goodell

Kearney Agricultural Center, University of California, Davis, California, U.S.A.

INTRODUCTION

Lygus bugs are pests of many crops including cotton, tobacco, beans, seed crops, strawberries, fruit and nut crops, ornamentals, and vegetables. Reproductive structures including buds, flowers, and fruits are commonly attacked. Losses are incurred through yield reduction and decreased quality. Efforts to develop host plant resistance have not been successful,^[1] and biological control does not play a prominent role in lygus management.^[2] Thus control options for *Lygus* spp. are limited to insecticides.^[2] However, resistance to pyrethroids has increased in recent years and continues to intensify.^[3] In addition, cross resistance to organophosphates and carbamates has developed.^[3] The development of resistance, together with an increasing desire to reduce the pesticide load in the environment, has led to a search for alternative lygus management strategies. Among the most promising is the manipulation of harvesting in adjacent forage alfalfa.

LYGUS BUGS AND FORAGE ALFALFA

Forage alfalfa can tolerate large numbers of lygus bugs without sustaining injury. It is a crop preferred by lygus bugs and they build up to high numbers during the summer. When alfalfa is harvested, the lush, humid, and cool environment is transformed into a dry, hot setting. Immature lygus are killed, but adults emigrate to nearby crops where they can cause considerable damage.

MANIPULATING ALFALFA HARVEST TO MANAGE LYGUS BUGS

In the 1960s, Stern, van Den Bosch, and Leigh^[4] developed the idea of harvesting alfalfa in alternate strips so that some lush alfalfa would always be present in the field. When one set of strips was cut, the

alternate strips were about half grown and the field was never completely devoid of lush alfalfa. Lygus bugs moving out of the cut strips flew into the uncut strips, rather than leave the field. Analysis of lygus bug populations in strip cut fields showed that emigration was significantly reduced.^[4,5] In addition, lygus nymphs from eggs laid by adults that moved into the uncut strips did not have time to mature before these strips were cut, and they were killed by exposure to unfavorable conditions.^[4] Strip cutting has other advantages as well. Populations of predators including big-eyed bugs, nabid bugs, minute pirate bugs, green lacewing, and ladybird beetles are all favored by strip cutting.^[6] *Aphidius smithi*, a parasite of the pea aphid, and parasites of lepidopterous pests, particularly alfalfa caterpillar, are conserved by strip cutting.^[6] Unfortunately, strip cutting poses certain problems.^[6] The two strips must be farmed as though they were separate fields with regards to irrigating and harvesting.^[4] This causes irrigation and equipment scheduling problems to which custom harvesters are unsympathetic. For these reasons, strip cutting has not been widely adopted by growers.^[6]

Summers^[7] proposed an alternative harvesting strategy termed "border cutting" to overcome the problems associated with strip cutting. Border cutting provides the same stable environment within the alfalfa ecosystem as does strip cutting and reduces the emigration of insects, including lygus, from alfalfa to adjacent crops. This technique consists of leaving approximately 10 ft of uncut alfalfa on alternate irrigation borders across the field. At the next cutting, these strips were cut, whereas uncut strips were left on alternate borders. This technique worked well for retaining natural enemies in the field. The number of entomophagous species recovered in the border cut fields was 2.5 times that collected in solid cut fields. Border cutting also retained approximately three times the number of adult lygus bugs compared with solid cutting. At each cutting, the alfalfa left standing at the previous harvest is split, with 50% deposited into the windrow to the right of the levee and 50%

deposited into the windrow on the left. This technique blended approximately one third “old” hay with two thirds “new” hay to minimize quality problems. No differences in quality were found between alfalfa harvested from the border cut fields and those from solid cut fields. Border cutting offers advantages over strip harvesting. First, except for the alternate strips on the levees, the entire field is cut at one time, alleviating scheduling problems. Second, the uncut strips do not interfere with irrigation.

Although these two strategies accomplish the same basic goal, they differ in the amount of alfalfa left uncut at each harvest. Strip cutting retains 50% of the alfalfa uncut at any one time, whereas border cutting retains approximately 10%. We conducted studies to determine the within-field movement of lygus bugs, the optimum amount of alfalfa to be left uncut to manage lygus bugs efficiently, and the most efficacious configuration of uncut strips. We evaluated the appropriate blending ratio of “old” to “new” hay required to alleviate quality problems.

WITHIN-FIELD MOVEMENT OF LYGUS

It has been suggested that lygus adults are “herded” across an alfalfa field in advance of the swather. However, we found no evidence of an increasing lygus “front” because of “herding” of lygus in advance of the swather (Fig. 1). Adults tend to fly straight up in front of the swather and then immediately return to the cut swath as it is expelled through the conditioner. There is no tendency for the adults to move either upwind or down wind from the swather. As the cut swath dries, the adults move into the uncut strips.

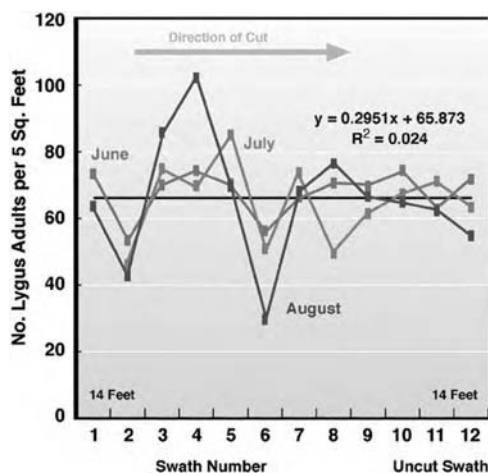


Fig. 1 Movement of lygus bugs across an alfalfa field in advance of the swather during three separate cuttings.

WITHIN-FIELD CONFIGURATION AND AMOUNT OF UNCUT ALFALFA

A single uncut block of alfalfa, comprising either 2.5% or 10% of the field, was left uncut at each harvest. Cotton was planted on each side of the field to evaluate lygus movement following the alfalfa harvest (Fig. 2). Using a D-vac, lygus adults were sampled in the alfalfa and cotton prior to cutting and again at 4, 8, 24, and 48 hr after cutting. There were no differences in lygus populations between the 2.5% and 10% uncut blocks of alfalfa (Fig. 3). The majority of lygus bugs remained in the uncut alfalfa blocks and lygus populations in the cotton remained below critical treatment thresholds^[8] (Fig. 4).

To understand lygus movement within the alfalfa field and into the cotton, lygus were marked with vertebrate proteins and released back into the alfalfa the same day.^[9] Lygus were collected from the uncut strips 4, 8, 24, and 48 hr after harvest and evaluated using enzyme-linked immunosorbent assay (ELISA).^[9] There was some movement of lygus between the 2.5% and 10% strips, but no more than 8% of the population moved in either direction. Lygus adults from the 2.5% and 10% strips were recaptured in the adjacent cotton, but no marked bugs from the 2.5% strips were captured in cotton adjacent to the 10% strip and vice versa, indicating only limited movement.

In another study, uncut alfalfa strips again composed 2.5% or 10% of the field. However, rather than a single block on the edge of the field, the equivalent amount of uncut alfalfa was left in a series of strips across the field. In addition to cotton, blackeye beans were planted adjacent to the alfalfa (Fig. 5). Both 2.5% and 10% of the uncut alfalfa strips retained equal numbers of lygus (Fig. 6). More lygus bugs were collected in the alfalfa strips than either the cotton or beans. Lygus numbers were higher in the beans than

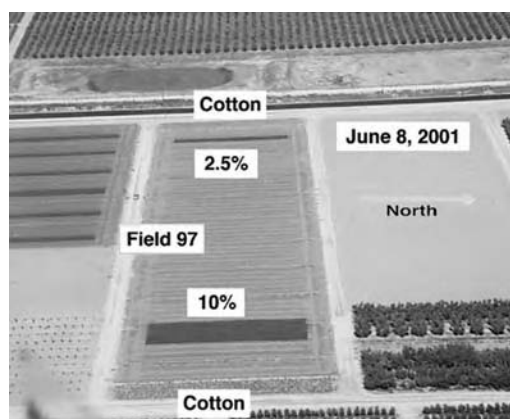


Fig. 2 Configuration of uncut alfalfa strips and sentinel cotton.

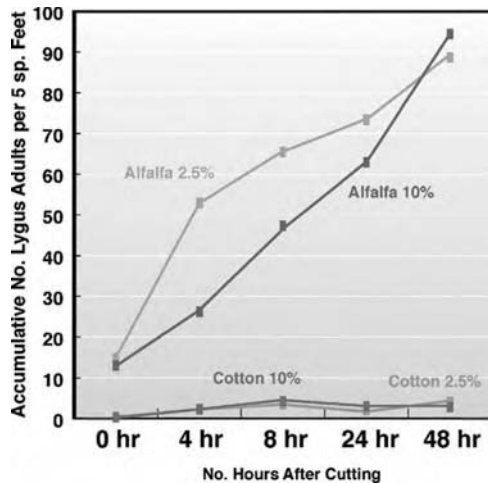


Fig. 3 Movement of lygus bugs into uncut alfalfa and cotton in a field in which 2.5% and 10% of the alfalfa were left uncut as a large block on one edge of the field.

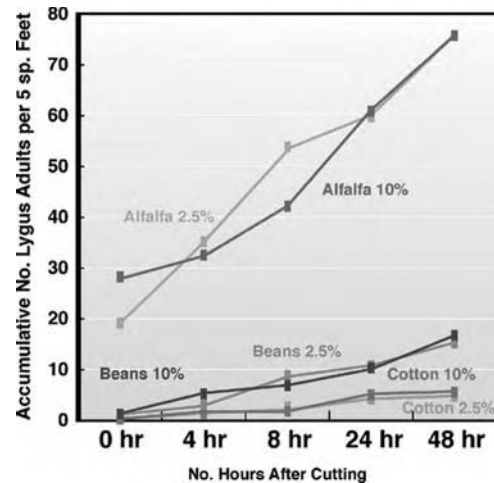


Fig. 5 Movement of lygus bugs into uncut alfalfa, cotton, and beans in a field in which 2.5% and 10% of the alfalfa were left uncut in small strips across the field.

in the cotton, but were still below threshold levels in both.^[8,10]

ALFALFA QUALITY

Bales containing various blends of old and new alfalfa were evaluated for quality including crude protein (CP), dry matter (DM), and acid detergent fiber (ADF). Total digestible nutrients (TDNs) and net energy of lactation (NEL) were calculated. Visual inspection by a qualified alfalfa broker was also conducted. Laboratory and visual inspection showed differences in quality between bales containing 100% new hay, 50%:50% new/old hay, and 25%:75% new/

old hay. There was no visual or chemical difference between 93%:7% new/old alfalfa and 100% new alfalfa.

CONCLUSION

Leaving uncut strips in alfalfa can reduce the movement of lygus into neighboring crops. Leaving at least some uncut alfalfa at each harvest appears to be more important than the quantity left. Leaving several strips across the field is preferable to leaving a single block of uncut alfalfa. However, the effect on hay quality must be considered. Some blending is acceptable without loss of quality.

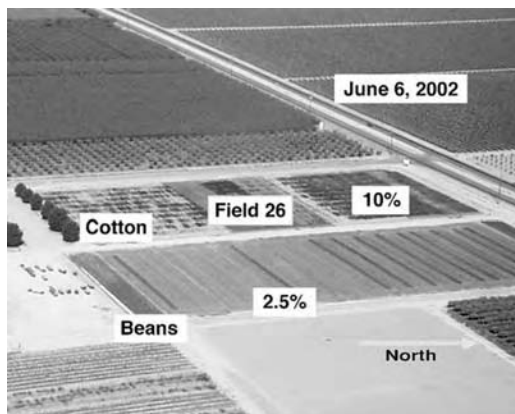


Fig. 4 Configuration of uncut alfalfa strips, sentinel cotton, and beans. Sentinel cotton and beans were also planted on the north edge of the field (not visible in photograph) in reverse order of the planting on the south.

REFERENCES

1. Teuber, L.R.; Labavitch, J.M.; Shackel, K.A.; Greve, C.E.; Taggard, K.L.; Gibbs, L.K. Approaches to Improving Plant Resistance to Lygus, Lygus Summit, University of California Lygus Conference, Visalia, CA, Nov 28, 1999, 78–81.
2. Ruberson, J.R.; Williams, L.H. Biological control of *Lygus* spp.: a component of areawide management. Southwest. Entomol. Suppl. **2000**, *23*, 96–110.
3. Grafton-Cardwell, B.; Christiansen, J.; Striggow, B.; Montez, G.; Brindley, B. Insecticide resistance trends in San Joaquin Valley lygus bugs, 1999. http://www.uckac.edu/cottonipm/PDF_files/summit_papers/bethgc.pdf (accessed July 2003).
4. Stern, V.M.; van Den Bosch, R.; Leigh, T.F. Strip cutting alfalfa for lygus bug control. Calif. Agric. **1964**, *18* (4), 5–6.

5. Stern, V.M.; van Den Bosch, R.; Leigh, T.F.; McCutcheon, O.D.; Sallee, W.R.; Houston, C.E.; Garber, M.J. *Lygus Control by Strip Cutting Alfalfa*; University of California Agricultural Extension Service, 1967; 1-13AXT-241.
6. van Den Bosch, R.; Stern, V.M. The Effect of Harvesting Practices on Insect Populations in Alfalfa, Proceedings of the Tall Timbers Conference on Ecological Animal Control by Habitat Management. No. 1. Tall Timbers Conference, Tallahassee, FL, Feb, 27-28, 1969; Tall Timbers Research Station: Tallahassee, FL, 1969, 47-54.
7. Summers, C.G. Population fluctuations of selected arthropods in alfalfa: influence of two harvesting practices. *Environ. Entomol.* **1976**, *5*, 103-110.
8. Godfrey, L.D.; Goodell, P.B.; Grafton-Cardwell, E.; Toscano, N.C.; Natwick, E.T. Insects and mites. In *UC IPM Pest Management Guidelines: Cotton*; UC ANR Publication 3444, 2002. <http://www.ipm.ucdavis.edu/PMG/r114301611.html> (accessed July 2003).
9. Hagler, J.R.; Cohen, A.; Bradley-Donlop, D.; Enriquez, F.J. New approach to mark insects for feeding and dispersal studies. *Environ. Entomol.* **1992**, *21*, 896-900.
10. Godfrey, L.D. Insects and mites. In *UC IPM Pest Management Guidelines: Dry Beans*; UC ANR Publication 3446, 2002. <http://www.ipm.ucdavis.edu/PMG/r52301211.html> (accessed July 2003).

Mammal Trapping

Gilbert Proulx

Alpha Wildlife Research and Management Ltd., Sherwood Park, Alberta, Canada

INTRODUCTION

Traps are mechanical devices used to capture animals. Trapping often is the most efficient way to selectively remove nuisance animals or reduce rodent densities in urban settings. In agriculture and forestry, trapping is a valuable alternative to non-selective toxicants. Every year, millions of rodents and carnivores are trapped for damage and disease control, and population regulation. This entry reviews trap types and factors that affect their performance, trapping strategies and concerns, and future needs.

TRAP TYPES

Mammal traps can be classified as killing or restraining mechanical devices. Killing traps consist of one or more striking jaws (or snare noose) activated by one or many springs upon firing of a trigger mechanism. Killing traps vary in size and mode of action (Fig. 1). Mousetrap-type devices, where one jaw closes 180° upon a flat surface, are most commonly used for the capture of commensal rodents, i.e., rats (*Rattus* spp.) and mice (*Mus* spp. and *Peromyscus* spp.).^[1] Killing boxes, spear- and pincer-type traps, and various models of body-gripping devices are used to capture fossorial rodents (*Thomomys* spp. and *Spermophilus* spp.) and moles.^[2] Planar traps, where a spring acts as a killing bar, are used to control rat-size rodents and small carnivores (e.g., weasel family). Rotating-jaw (Conibear-type) traps with a scissor-like closing action are used for a variety of animals ranging from tree squirrels (*Tamiasciurus* spp. and *Sciurus* spp.) to beaver (*Castor canadensis*). Finally, manual locking and power snares are used to kill larger animals such as red fox (*Vulpes vulpes*) and coyote (*Canis latrans*).^[1]

Restraining traps are devices designed to capture an animal alive (Fig. 1). Three main types are used in the control of mammal pests. Cage/box traps are produced in a variety of sizes for small insectivores and rodents, carnivores, and ungulates.^[1] They are made of wire or nylon mesh, plastic, or wood. The functional parts of these traps include the cage/box, one or two self-closing doors, a door lock mechanism, a trigger,

and a treadle or trip pan. Foothold traps are commonly used to capture medium-size animals such as coyote and fox. Typically, these traps consist of two jaws open at 180° at set position, and closing 90° upon each other at firing time. Another foothold design is the EGG trap with a pull trigger that releases a small striking bar to block an animal's paw, and a plastic housing that protects the captured limb from torsion injuries. This trap is specifically used for the capture of raccoons (*Procyon lotor*) and Virginia opossum (*Didelphis virginiana*).^[3] Finally, foot snares are spring-powered cables used to capture medium- and large-size mammals.

TRAP EFFICIENCY

Trap efficiency, which is the rate at which a trap catches the intended species, varies greatly within and between years. Factors affecting trap efficiency relate to trapping methods, environmental variables, and biological variables.^[4] Trap types, sets, and sites must be carefully selected for target species. However, the number of trapping devices deployed and the selection of bait or lure significantly affect trap performance. Meat, fatty substances, seeds, vegetables, fruits, nuts, and scented lures (conspecific odors or food-related scents) usually increase trapping success. Bait efficiency may vary seasonally due to differences in animals' activity patterns and natural food availability. Prebaiting, where trapping sites or traps themselves are rendered inoperative and baited, is often recommended to effectively remove pest animals. Weather conditions may impact the operation of trapping devices and the behavior of target species. Finally, population density and distribution, animal movements, and the individual response of animals to traps vary greatly between areas and over time.

TRAPPING STRATEGIES AND CONCERNS

The efficiency and costs of mammal trapping control programs are difficult to estimate because of the above-noted factors, the number and experience of trappers, and the goal of a particular pest control

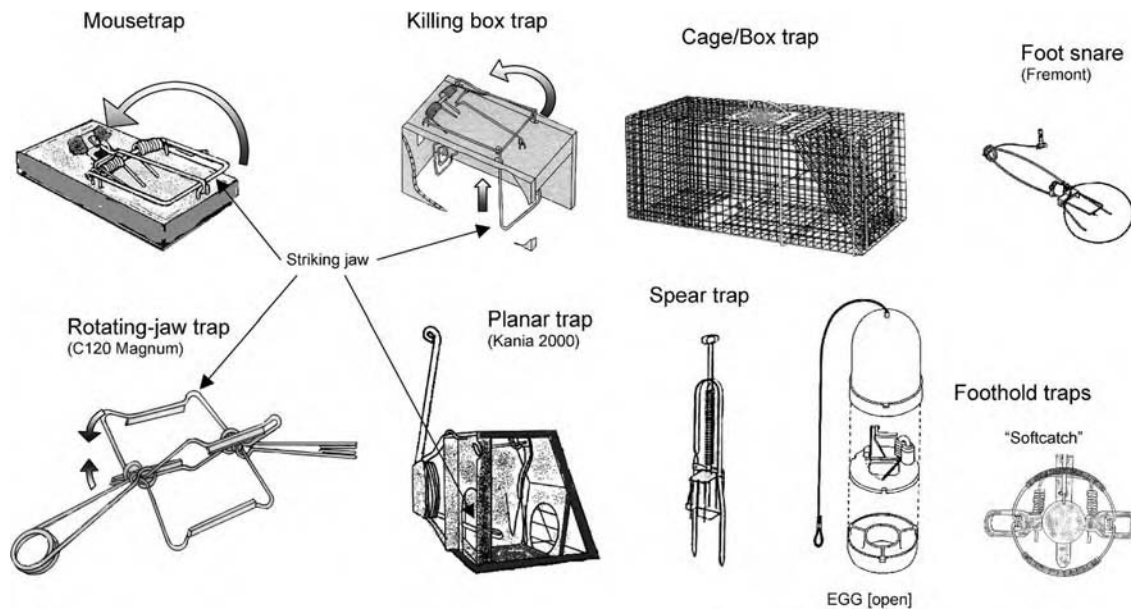


Fig. 1 Diagrams of killing and restraining traps used to capture mammal pests.

program. Ideally, control trapping should be conducted before pest populations reach high-density levels, e.g., before the birth of young of the year. Eradication through trapping is seldom achievable except on a local scale, and usually at high cost.^[5] Sporadic or occasional control is ineffective, as pest numbers usually return to precontrol levels soon after the trapping effort. Sustained control is far more cost-effective as it involves a reduction of populations to low levels and ongoing maintenance (often involving a control buffer zone) to minimize reproduction and immigration.^[6-9] This is a “preventive” strategy that is particularly effective to control rodents. A variant of the sustained control strategy is the removal of a small proportion of the population causing the impact. This is a “corrective” strategy often used in the case of wild canids and other large carnivores preying on livestock.^[10]

There are growing concerns about trap selectivity and the welfare of mammals, pests included.^[1,6] Effective techniques have been developed for avoiding the capture of non-target species. Responsible trapping is facilitated with the use of restraining traps, which allow one to release unwanted animals and to remove specific individuals. Restraining traps should hold animals with minimal distress and trauma. They should be checked daily, and captured animals should be immediately relocated, released, or euthanized. Killing traps should render animals irreversibly unconscious as quickly as possible. They should be used when there is no risk of injury for humans and domestic animals.^[11]

FUTURE NEEDS

Although trapping plays an important role in control programs, little work has been conducted on mammal pest traps from an efficiency, selectivity, and animal welfare point of view. There is a need for trap research and development for the control of commensal rodents, and new trap alternatives for medium- and large-size carnivores.^[1] Future efforts aimed at improving animal handling, understanding factors that impact trap performance, and integrating trapping into pest management programs using various control methods should be promoted.

REFERENCES

1. Proulx, G. Review of current mammal trap technology in North America. In *Mammal Trapping*; Proulx, G., Ed.; Alpha Wildlife Research and Management Ltd.: Sherwood Park, Alberta, 1999; 1–46.
2. Marsh, R.E. *Pocket Gopher Traps: A Collector's Manual*; R.E. Marsh Publisher: Davis, CA, 1997.
3. Hubert, G.F., Jr.; Wollenberg, G.K.; Hungerford, L.L.; Bluett, R.D. Evaluation of injuries to virginia opossums (*Didelphis virginiana*) captured in the EGG trap. *Wildl. Soc. Bull.* **1999**, *27* (2), 301–305.
4. Pawlina, I.M.; Proulx, G. Factors affecting trap efficiency: a review. In *Mammal Trapping*; Alpha Wildlife Research and Management Ltd.: Sherwood Park, Alberta, 1999; 95–115.
5. Voigt, D.R.; Tinline, R.L. Fox rabies and trapping: A study of disease and fur harvest interaction. In *Midwest Furbearer Management*, Proceedings Symposium 43rd

- Midwest Fish and Wildlife Conference, Sanderson, G.C., Ed.; Wichita, KS, 1982, 139–156.
6. Braysher, M. *Managing Vertebrate Pests: Principles and Strategies*; Bureau of Resource Sciences, Australian Government Publishing Service, Canberra, 1993.
 7. Alberta Agriculture. In *A History of Rat Control in Alberta*; Agdex 682-1: Alberta Agriculture, Edmonton, 1989.
 8. Stroud, D.C. Dispersal and some implications for control of the California ground squirrel. In *Proceedings Vertebrate Pest Conference*; Marsh, R.E., Ed.; University of California: Davis, CA, 1982; Vol. 10, 210–213.
 9. Proulx, G. A Northern pocket gopher (*Thomomys talpoides*) border control strategy: promising approach. *Crop Prot.* **1997**, *16* (3), 279–284.
 10. Sacks, B.N.; Blejwas, K.M.; Jaeger, M.M. Relative vulnerability of coyotes to removal methods on a Northern California ranch. *J. Wildl. Manage.* **1999**, *63* (3), 939–949.
 11. Proulx, G.; Barrett, M.W. Animal welfare concerns and wildlife trapping: Ethics, standards and commitments. In *Transactions of the Western Section of the Wildlife Society*; Loft, E.R., Callas, R.L., Eds.; 1989; Vol. 2, 1–6.

Mammalia Pest Impacts in New Zealand

Phil Cowan

*Department of Vertebrate Pest Ecology, Landcare Research New Zealand, Ltd.,
Palmerston North, New Zealand*

INTRODUCTION

Most mammal pest control is carried out to reduce losses to agricultural production resulting from either damage to crops or transmission of parasites and diseases to livestock or people. However, in some countries, such as New Zealand and Australia, there are equally serious conservation problems caused by the impacts of introduced mammals on indigenous plants and animals.

INTRODUCED MAMMAL PESTS

New Zealand is uniquely vulnerable to mammal pests, having no indigenous mammals except bats and marine mammals, an insular fauna and flora isolated for some 80 million yr, and the highest number of introduced mammal species of any country (Fig. 1).^[1,2] Its major introduced mammal pests are the Australian brushtail possum, a suite of ungulates (four species of wild deer, feral pig, feral goat, chamois, Himalayan tahr), a suite of carnivores (feral cat, feral ferret, stoat, weasel), and a suite of rodents and lagomorphs (European rabbit, European hare, brown rat, black rat, Polynesian rat, house mouse).^[2] The impacts of these pests, the priority for their control, and the various control methods have been summarized.^[3]

CONSERVATION IMPACTS

In New Zealand, introduced browsing or grazing mammals affect indigenous ecosystems at all levels. The brushtail possum is directly responsible for major canopy damage and change in native forest;^[4] the ungulates, lagomorphs, and rodents change understory composition and prevent regeneration in forest and non-forest habitats,^[2,5] and all contribute to erosion and exotic weed invasion problems in indigenous ecosystems.^[6] The combined impacts of the suite of browsers/grazers are sufficient to exterminate palatable species locally and to threaten some rare species with extinction.^[4,7]

The introduced rodents and carnivores are serious predators of native animals, including invertebrates such as large land snails.^[2] Populations of NZ iconic

bird species, such as kiwi and kokako, are in rapid decline^[8,9] and one rare species, the flightless kakapo parrot, only survives because it has been translocated to predator-free islands. Increasingly, the importance of the omnivorous brushtail possum as a predator is being recognized.^[10]

Complex ecological interactions between native and introduced species may exacerbate impacts. For example, mast seeding of native beech trees is followed by eruptions of introduced house mice, and later by increases in introduced stoats (which prey on mice). As mouse numbers decline, stoat predation on some native birds increases.^[11] Control of feral cats may result in increases in introduced rodent numbers so that any benefit from reduced cat predation on native animals may be offset by increased rodent predation on the same or other species. Browsing by introduced possums suppresses flowering and fruiting of some native species, leading to potential food shortages for native animals.^[12]

MANAGEMENT APPROACHES

New Zealand government agencies spend more than NZ\$71 million annually on pest and weed control for conservation.^[13] Most animal control is directed at the brushtail possum. In 1999/2000, the NZ Department of Conservation (DoC) spent about NZ\$15 million controlling possums, NZ\$5 million on goats, and NZ\$8 million on other pests. DoC aims to sustain the current level of 1.1 million ha under possum control to prevent canopy collapse and species loss, and eventually to increase this level to cover the 1.8 million ha of highest priority ecosystems identified in the National Possum Control Plan, which set out goals and targets over a 10-yr time frame. Areas for possum control are ranked using a set of criteria that primarily take into account the conservation value of plants and animals found there, and their vulnerability to possums. Eradication proposals must meet further criteria relating to feasibility.^[14] Similar National Control Plans and ranking systems operate for deer and goats. For the other main mammal pests, most control is centered on offshore islands, where there have been an increasing number of successful eradication programs

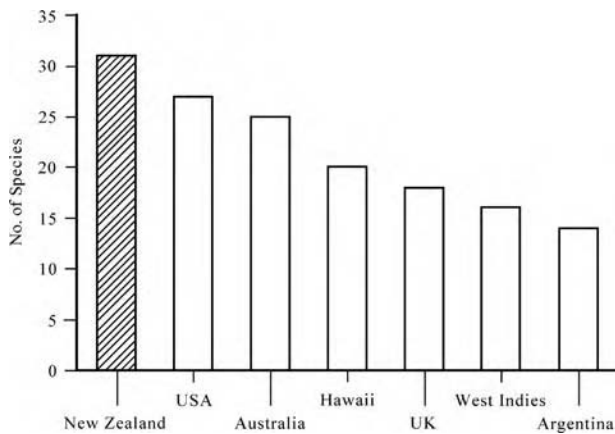


Fig. 1 Numbers of mammals introduced to various countries.

for rodents, lagomorphs, and carnivores (mainly feral cats and stoats), and on mainland “islands.” Mainland islands are usually isolated patches of 500–5000 ha of high conservation value where intensive control of all introduced mammal pests is undertaken on a sustained basis.^[9] The conservation benefits of such an approach are great, but so is the cost.

One of the major problems in control of introduced mammals to protect native plants and animals is in establishing targets for control—that is, to what levels do numbers of introduced mammals need to be reduced to protect or allow recovery of native species or ecosystems?^[15] Unlike management of agricultural or disease problems, there is unlikely to be a single target or threshold pest density to protect conservation values, particularly where impacts are on native plants. Some species, such as native mistletoe, continue to be severely browsed even at very low possum densities, whereas other species are much less susceptible.^[4,12] Thus, to maintain an intact canopy in a forest may only require reduction of possum numbers by 60–70%, but to retain mistletoe in that forest may require possum numbers to be reduced by 95% or more. The application and use of a bioeconomic framework for pest management could allow a number of these issues to be resolved.^[16]

CURRENT AND FUTURE TOOLS

Introduced mammals in New Zealand are controlled principally with a variety of toxins, leg-hold and kill traps, and by DoC-organized, recreational, and commercial hunting. Toxins are used primarily for possum, rodent, lagomorph, and carnivore control, while hunting focuses on deer, pigs, and goats. The most commonly used toxin is 1080 poison (sodium monofluoroacetate), and New Zealand is the world’s largest user (c. 2 t in 1998/1999). In recent years, a number of

alternative toxins, particularly anticoagulants, such as brodifacoum and pindone, have been approved for possum, rodent, and rabbit control. Over the last 10 yr, the efficacy of many control tools has increased significantly,^[17] and a number of new approaches, such as encapsulation of toxins to avoid problems of bait and poison shyness and the use of global positioning systems to ensure even flight coverage during aerial poisoning, have been adopted. New and improved techniques for operational monitoring (success at reducing pest numbers) and performance monitoring (success at reducing pest impacts) have also been developed.

Because of the widespread nature of some of the mammal pests in New Zealand, a limited budget for mammal pest management for conservation, and difficulties with control using current technologies, research is currently underway to develop biological control based on interfering with fertility.^[18] Initially, this is focussed on possums, but an evaluation of the technology for stoat control was completed recently and some preliminary research begun. The general approach is to develop immunologically based contraception or sterility, similar to the approaches being taken in Australia for fox, rabbit, and mouse control. The use of hormone–toxin complexes that would target gonadotrophin-releasing hormone (GnRH) producing cells in the hypothalamus is also being investigated. Initially, fertility control for possums is likely to be bait-delivered, but research is also underway to assess the potential of possum-specific viruses and nematode parasites for transmissible delivery of fertility control. Fertility control is likely to be used with conventional control to slow the rate of recovery of pest numbers, and hence reduce the frequency of control, with concomitant cost savings and reductions in toxin use, risks to non-target species, and environmental contamination. Research into public attitudes to mammal pest management in New Zealand indicates support for this approach.^[19]

REFERENCES

1. Lever, C. *Naturalised Mammals of the World*; Longman: London, 1985.
2. King, C.M., Ed.; *The Handbook of New Zealand Mammals*; Oxford University Press: Auckland, 1990.
3. Cowan, P.E.; Tyndale-Biscoe, C.H. Australian and New Zealand mammal species considered to be pests or problems. *Reprod., Fertil. Dev.* **1997**, *9* (1), 27–36.
4. Payton, I. Damage to native forests. In *The Brushtail Possum*; Montague, T.L., Ed.; Manaaki Whenua Press: Lincoln, New Zealand, 2000; 111–125.
5. O’Donnell, C.F.J. *Possums as Conservation Pests*, Proceedings of a National Science Strategy Committee Workshop, Wellington, New Zealand, Nov 29–30,

- 1994; Department of Conservation: Wellington, New Zealand, 1995.
6. Cowan, P.E. Environmental pests: how can we identify the values at risk? *N.Z.J. Zool.* **1993**, *20*, 279–283.
 7. Campbell, D.J. Changes in structure and composition of a New Zealand lowland forest inhabited by brushtail possums. *Pac. Sci.* **1990**, *44*, 277–296.
 8. McLennan, J. *Ecology of Brown Kiwi and Causes of Population Decline in Lake Waikaremoana Catchment*; Conservation Advisory Science Notes 167; Department of Conservation: Wellington, New Zealand, 1997; 1–25.
 9. Innes, J.; Hay, R.; Flux, I.; Bradfield, P.; Speed, H.; Jansen, P. Successful recovery of North Island kokako *Callaeas cinerea wilsoni* populations, by adaptive management. *Biol. Conserv.* **1999**, *87*, 201–214.
 10. Sadleir, R. Evidence of possums as predators of native animals. In *The Brushtail Possum*; Montague, T.L., Ed.; Manaaki Whenua Press: Lincoln, New Zealand, 2000; 126–131.
 11. O'Donnell, C.F.J.; Phillipson, S.M. Predicting the incidence of mohua predation from the seedfall, mouse, and predator fluctuations in beech forests. *N. Z. J. Zool.* **1996**, *23* (3), 287–293.
 12. Nugent, G.; Sweetapple, P.; Coleman, J.; Suisted, P. Possum feeding patterns: dietary tactics of a reluctant Folivore. In *The Brushtail Possum*; Montague, T.L., Ed.; Manaaki Whenua Press: Lincoln, New Zealand, 2000; 10–19.
 13. Hackwell, K.; Bertram, G. *Pests and Weeds The Cost of Restoring the Dawn Chorus*; New Zealand Conservation Authority: Wellington, 1999.
 14. Department of Conservation. *National Possum Control Plan 1993–2002*; Department of Conservation: Wellington, 1994.
 15. Hickling, G.J. Possums as conservation pests. In *Action Thresholds and Target Densities for Possum Pest Management*, Proceedings of a National Science Strategy Committee Workshop, Wellington, New Zealand, Nov 29–30, 1994; O'Donnell, C.F.J., Ed.; Department of Conservation: Wellington, 1995; 47–52.
 16. Choquenot, D. A bioeconomic framework for pest control in New Zealand, [Available at www.landcare.cri.nz/conferences/manaakiwhenua/papers/].
 17. Morgan, D.; Eason, C. Improving conventional approaches to possum control. In *Possum/Bovine Tb NSSC Annual Report 1995/96*; Royal Society of New Zealand: Wellington, 1996; 16–20.
 18. Cowan, P. Biological control of possums: prospects for the future. In *The Brushtail Possum*; Montague, T.L., Ed.; Manaaki Whenua Press: Lincoln, New Zealand, 2000; 262–270.
 19. Fitzgerald, G.; Wilkinson, R.; Saunders, L. Public Perceptions and Issues in Possum Control. In *The Brushtail Possum*; Montague, T.L., Ed.; Manaaki Whenua Press: Lincoln, New Zealand, 2000; 187–197.

Mass-Trapping

Masashi Kakizaki

*Hokkaido Ornamental Plants and Vegetables Research Center,
Takikawa, Hokkaido, Japan*

INTRODUCTION

The idea of insect pest control by mass-trapping is a simple one. Populations of target insect pests are reduced by capturing many individuals in them using many traps baited with a species-specific attractant. For sex pheromone-based mass-trapping, the rate of female copulation is reduced because of male annihilation, the density of fertile eggs laid decreases, and the population of the next generation is smaller. Although many males might visually be captured by sex pheromone traps, a high proportion of individuals in a population must be captured for pest control by mass trapping. Various types of traps and lure-and-kill formulations have been used for mass-trapping.

ATTRACTANT SOURCE

The attractants used include semiochemicals (sex pheromones, aggregation pheromones, kairomones), food odors, other synthetic attractants, light sources, and colors. For the source to be effective, it must be more attractive than natural attractants, for example, females or food.

TRAPS

Traps must be highly efficient, with large capacity, because of their use during extended trapping periods covering the occurrence season of the target insect. They also need to be inexpensive and easy to set and maintain. Many kinds of traps are used for target insects because their design must match the approaching behavior of the particular insect targeted. A successful trap should capture a high proportion of the target insect and few non-target insects and/or small animals.

The types of traps used are sticky board traps (Pherocon 1C wing trap, Delta trap, Takeda-shiki trap, Jackson trap, etc.), water-pan traps, funnel traps, liquid traps (McPhail trap), dry traps (Nadel trap, Steiner trap, Takeda-shiki box trap, Tephri trap), net cage traps, and other handmade traps (Pet-bottle trap, box traps, etc.).

LURE AND KILL

Lure-and-kill-type formulations contain attractants and insecticides. Insects are attracted to the lure and are killed after touching or eating it. By scattering or setting the formulations, a whole field or area can be covered. This type of formulation is not saturated for capturing of insects.

The types of formulations used are as follows:

- stick tube formulation (stick coated with pheromone (Ph) and insecticide (In)) used for the cotton boll weevil;
- fiberboard formulation (sugarcane fiberboard impregnated with Ph and In) used for the sweet potato weevil, the yellowish elongate chafer and the oriental fruit fly;
- micro-capsule spray (pheromone is encapsulated in polyurea capsules or polymer beads and mixed with In) used for the olive fruit fly;
- ceramic tip (black-colored ceramic beads impregnated with Ph and In, which visually look like a female for males) used for the sweet potato weevil;
- target (net cage trap sprayed with In and set with attractant) used for the tsetse flies;
- toxic bait (diet contained attractant and In) used for flies, etc.

TRAP DENSITY, AREA SIZE IN TREATMENT, AND FACTORS FOR CONTROL

It is important to establish optimal trap densities and the minimum area necessary for effective pest control. This may mean analyzing the lure attraction range, adult flight range, and immigration from non-treatment areas. However, it is difficult to investigate these parameters for each insect and many experiments have been done to evaluate the various trap densities and treatment area sizes tested. For female sex pheromone-based mass-trapping, the trap densities are generally lower (e.g., 0.2–1 traps/ha) because of the long distance of adult mating flights and a large quantity of attraction by lure, whereas they tend to be high if the distance of adult mating flights is short. Treatment areas would need to be large for species in which many

adults migrate from non-treated areas and for polyphagous species. In contrast, small treatment areas may be possible if there is little migration of adults and if the pest is a monophagous species. It is also important that males are captured before mating (for female sex pheromone) or that females are caught before they could lay eggs (for aggregation pheromone). Species that have males occurring earlier than females or are present only for a short period may be suitable for male annihilation.

THEORETICAL EXPERIMENTS

To control some species of Lepidoptera by mass trapping, Knipling and McGuire^[1] reported that the male catch must be as high as 80–95%, and Roelofs et al.^[2] estimated that five traps per female are needed for a 95% reduction in female fecundity. Nakasuji and Fujita^[3] showed the relation between the effect of mass-trapping depending on population density and the mating probability using a simulation model.

EXPERIMENTS ON PHEROMONE-BASED MASS-TRAPPING

Pheromone-based mass-trapping was tested for more than 58 species (39 Lepidoptera, 18 Coleoptera, and 1 Diptera). Those evaluated in the field are shown in Table 1. Experiments were performed on trap densities of 100 traps/10a to 0.2 traps/ha in areas of 120 m² to 6287 ha. The kinds of trap used in these experiments were water-pan, funnel and sticky board traps, and fiberboard formulations. The effects on insect pests included reductions in female copulation rate, population density, injury, yield loss, and reduced insecticide applications. Control effects of mass-trapping are clear at low densities or in the early part of an insect pest season. However, they are often less clear at high densities. Although some insect pests that are difficult to control by insecticides are listed in Table 1, mass-trapping was available for these.

PRACTICAL USE OF PHEROMONE-BASED MASS-TRAPPING

For the cotton boll weevil, *Anthonomus grandis* Both. (Coleoptera), male product aggregation pheromone, (+)-(Z)-2-isopropenyl-1-methylcyclobutane-ethanol, (Z)-3,3-dimethyl- $\Delta^{1,\beta}$ -cyclohexaneethanol, (Z)-3,3-dimethyl- $\Delta^{1,\alpha}$ -cyclohexaneacetaldehyde, (E)-3,3-dimethyl- $\Delta^{1,\alpha}$ -cyclobutaneacetaldehyde, attracts females in summer, and both sexes during overwintering.^[4] Lure-and-kill-type formulations 'Grandlure' and insecticides, BWACT (Boll Weevil Attract and Control Tube),

TMP (Tubo Mata Picudo), or TMB (Tubo Mata Bicudo) were set 1/ha in 20,000 ha (Bolivia) to 6,000,000 ha (USA) of cotton fields. IPM programs that used these had reduced boll weevil populations and insecticide usage and increased cotton yields. Further, *A. grandis* has been eradicated in California, southwestern Arizona, and a part of Mexico, and IPM programs for eradication are underway in the USA and Latin America (Mexico, Argentina, Bolivia, Brazil, Colombia, Paraguay).

For the sweet potato weevil, *Cylas formicarius* (Fabricius) (Coleoptera), lure-and-kill-type fiberboard formulation 'Sweetbilure' ('Arimodoki call': female sex pheromone (Z)-3-dodecenyl (E)-2-butenolate and insecticide) was set 4–25/ha in and around sweet potato fields, and the densities of *C. formicarius* males and the mating ratio of females decreased in the treatment area (Japan, India).

For the sugarcane wireworms, *Melanous okinawensis* Ohira and *M. sakishimensis* Ohira (Coleoptera), water-pan or funnel-vane traps baited with the lures 'Okimeranolure' ('Okinomera call': *n*-dodecyl acetate) and 'Sakimeranolure' ('Sakinomera call': (E)-9,11-dodecadienyl butyrate and (E)-9,11-dodecadienyl hexanoate) are usually set 1–1.5/ha. When 120 traps were set in 25 ha sugarcane fields, there was a 30–40% reduction in sugarcane wireworm population densities of the former species (Japan).

For the noctuid, *Spodoptera litura* (Fabricius) and *S. littoralis* (F.) (Lepidoptera), traps baited with the lure 'Litlure' ('Pherodine SL': (Z,E)-9,11-tetradecadienyl acetate and (Z,E)-9,12-tetradecadienyl acetate, or only (Z,E)-9,11-tetradecadienyl acetate) were set 1–100/ha in 10–20,000 ha of treatment area. For *S. litura*, the densities of larval colonies decreased at the beginning of occurrence (Japan), and examinations indicated a reduction in the use of insecticides sprays for *S. litura* and *S. littoralis* (USA, Israel, Japan, Crete, India, and Taiwan). However, when trap densities were high, the effects of mating disruption were found to be greater than those of mass-trapping. Other experiments with less effective control were reported for USA, Crete, UK, and Egypt.

USE OF OTHER SEMIOCHEMICALS

Mass-trapping using attractants from plants and their derivatives has also been conducted. Populations of the oriental fruit fly *Dacus dorsalis* (Diptera) have been successfully eradicated by the male annihilation method using the male attractant Methyl eugenol+ insecticides in the islands of Hawaii, Saipan, Mariana, Tenian, Amami, and Okinawa (USA, Japan). And 'Siglure' (6-methyl-3-cyclohexene-1-carboxylic acid 1-methyl-propyl ester), 'Medlure', and 'Trimedlure',

Table 1 Examinations of mass-trapping using pheromones and semiochemicals

| Species | Treatment | Effect ^a | Country |
|---|---|-----------------------|--|
| Lepidoptera | | | |
| <i>Adoxophyes</i> sp. small tea tortrix | 35 traps/720 m ² | E-IJ | Japan |
| <i>Argyrotaenia velutinana</i> redbanded leafroller | 12–25 traps/ha, 11 ha | E-IJ | USA |
| <i>Chilo suppressalis</i> | | NE | Japan, Philippines |
| <i>C. sacchariphagus indicus</i> | 25–37 traps/ha, 5–10 ha | E-IJ | India |
| <i>C. partellus</i> | | NE | Kenya |
| <i>Cydia funebrana</i> plum moth | 50 traps/ha | E-IJ | Romania |
| <i>C. molesta</i> oriental fruit moth | 15 traps/ha, 4000–5400 ha | E-C, IJ | USA, China |
| <i>C. pomonella</i> codling moth | 50 traps/ha or 1 trap/tree | E-IJ or NE | USA, India, Romania |
| <i>Epaestia elutella</i> warehouse moth | 14 traps/3300 m ² | E-D | |
| <i>Ephestia cautella</i> | | E-D | Italy |
| <i>E. kuehniella</i> | | E-IU | Israel |
| <i>Earias insulana</i> spiny bollworm | 4 traps/ha, 150 ha | E-IJ | Egypt, Syria |
| <i>Helicoverpa armigera</i> tomato fruit worm | 16–25 traps/ha | E-IJ | Taiwan |
| <i>Paralobesia viteana</i> grape berry moth | 12–25 traps/ha, 11 ha | E-IJ | USA |
| <i>Paranthrene regalis</i> grape clearwing moth | 53–55 traps/2.7 ha | E-C | China |
| <i>Pectinophora gossypiella</i> pink ballworm | 1–11 traps/ha, 3080–6287 ha | E-IU, D | Egypt |
| <i>Plutella xylostella</i> diamondback moth | 6 traps/field | E-IJ | India, Taiwan |
| <i>Plodia interpunctella</i> | | E-IU | Italy |
| <i>Rondotia meniciana</i> mulberry white caterpillar | 53 traps/3.3 ha | E-C, D | China |
| <i>Sesamia nonagrioides</i> | 10 traps/ha | E-IJ | Greece |
| <i>Spodoptera litura</i> tobacco caterpillar and <i>S. littoralis</i> cotton leafworm | 1–2 trap/ha, 20–20,000 ha, 52 traps/27 ha, 336 traps/590 ha | E-D, IU or NE | Egypt, Taiwan, Japan, UK, China, USA, India |
| <i>S. exigua</i> beet armyworm | 30 traps/ha, or 10 traps/vinyl house (330 m ²) | E-D, or E-D, IJ | Korea, Taiwan |
| <i>Synanthedon exitiosa</i> peach tree borer | 2.5–5 traps/ha | E-D | USA |
| <i>S. myopaeformis</i> | 10 trap/ha | E-IJ | Italy |
| <i>Thaumetopoea wilkinsoni</i> pine processionary caterpillar | 0.35–100 ha | E-D | Israel |
| Coleoptera | | | |
| <i>Anthonomus grandis</i> cotton boll weevil | 1 lure/ha, 20,000–60,000,000 ha | E-D, IJ, IU | USA, Argentina, Brazil, Colombia, Paraguay, Bolivia |
| <i>Cylas formicarius</i> sweet potato weevil | 1 trap/10 ha, 1–25 fiberboard/ha | E-D, IJ, or E-C, D | Japan, India, USA, Taiwan |
| <i>Heptophylla picea</i> yellowish elongate chafer | 100 fiberboards/10 a, 120–700 m ² | E-C, D | Japan |
| <i>Melanous okinawensis</i> and <i>M. sakishimensis</i> sugarcane wireworms | 1–1.5 traps/ha | E-IJ | Japan |
| Diptera | | | |
| <i>Dacus</i> (= <i>Bactrocera</i>) <i>oleae</i> | 0.266–2 traps/tree (use of sex pheromone, aggregation pheromone, food attractant) | E-IJ, IU | Greece |

^aEffect (E) or no effect (NE) to reduction of copulation (C), population density (D), injury (IJ), or insecticide use (IU).

their related compounds adducted hydrogen chloride, are effective against *Ceratitis capitata*, and 'Cue-lure' (4-(*p*-methoxy phenyl)-2-butanone) against the melon fly *D. cucurbita*, and *D. tryoni*. The bark beetles, *Ips paraconfusus*, *I. typographus*, *Scolytus multistriatus*, *Dendroctonus rufipennis*, *D. spp. Dryocoetes confusus*, are attracted to aggregation pheromones and the related monoterpenes: ipsenol, ipsdienol, (*Z*)-verbenol, *exo*-brevicomin, α -pinene, etc. Mass-trapping examinations using 530,000–600,000 traps captured 2900–4500 million beetles, and the population densities and injured level were reduced. However, these results indicated that many traps are necessary for control. The tsetse flies, *Glossina m. morsitans* and *G. pallidipes*, are attracted to a mixture of carbon dioxide, acetone, and 1-octen-3-ol, same as a natural ox odor. Treatment of the targets (net cage trap treated by insecticide) baited with these compounds decreased population density.

OTHER METHODS BY NON-SEMIOCHEMICALS

Other methods include mechanical mass-trapping methods. In the greenhouse and vinyl house, yellow, blue, and other colored sticky plate traps have been used for monitoring of whitefly and thrips. The control methods using these sticky traps, yellow-colored boards to white fly and pink-colored ribbons to western flower thrips, have been examined. By removing the diamond-back moth adults, *Plutella xylostella*, at intervals of 3 days to a week using an electro-vacuum cleaner in a greenhouse, the densities of *P. xylostella* population were reduced (Osaka Pref., Japan).

CONCLUSION

For mass-trapping to successfully control pests, it is necessary to match the trap density, type of lure and time of application to the type of insect; for example, using a high efficiency lure and trap, and catching a high proportion of individuals in a population before mating and/or oviposition. Because of these complexities, this method is not applied to as many insects as mating disruption methods. Although control by mass-trapping is effective at low densities or at the beginning of the occurrence of a pest infestation, it tended to fail when used for control at high densities. Therefore, this method should be used together with other control methods and with monitoring of a target insect population. Mass-trapping can be used with many other control methods: chemicals, cultural controls, and biological controls. In the future, applied mass-trapping should also be considered. Shapas, Burkholder, and Boush^[5] reported that population of the dermestid beetle, *Trogoderma glabrum*, was

suppressed by the introduction of entomophthorales, *Mattesia trogodermae*, by releasing males that were attracted to and touched a pheromone and their spores formulation. In the case of insect pests for which the direct effect of mass-trapping is not high, methods of attraction and infection might also be available. Then mass-trapping would be available as one of the control methods for IPM programs.

REFERENCES

1. Knipling, E.F.; McGuire, J.U., Jr. Population models to test theoretical effects of sex attractants used for insect control. U. S. Dept. Agric. Inf. Bull. **1966**, *308*, 1–20.
2. Roelofs, W.L.; Glass, E.H.; Tettel, J.; Comeau, A. Sex pheromone trapping for red-banded leaf roller control: Theoretical and actual. J. Econ. Entomol. **1970**, *63*, 1162–1167.
3. Nakasuji, F.; Fujita, K. A population model to assess the effect of sex pheromones on population suppression. Appl. Entomol. Zool. **1980**, *15*, 27–35.
4. Tumlinson, J.H.; Hardee, D.D.; Gueldner, R.C.; Thomson, A.C.; Hedin, P.A.; Minyard, J.P. Sex pheromones produced by male boll weevil: isolation, identification, and synthesis. Science **1969**, *166*, 1010–1012.
5. Shapas, T.J.; Burkholder, W.E.; Boush, G.M. Population suppression of *Trogoderma glabrum* by using pheromone luring for protozoan pathogen dissemination. J. Econ. Entomol. **1977**, *70*, 469–474.

BIBLIOGRAPHY

- Bakke, A.; Lie, R. Mass trapping (Chapter 4). In *Insect Pheromones in Plant Protection*; Jutsum, A.R., Gordon, R.F.S., Eds.; John Wiley & Sons: Chichester, 1989; 67–87.
- Campion, D.G., III. Mass-trapping. Survey of pheromone uses in pest control (Chapter 14). In *Techniques in Pheromone Research*; Hummel, H.E., Miller, T.A., Eds.; Springer-Verlag: New York, 1984; 420–427.
- Howse, P.; Stevens, I.; Jones, O. Mass-trapping (Chapter 10), and Lure and Kill (Chapter 11). In *Insect Pheromones and Use in Pest Management*; Chapman & Hall: London, 1998; 280–313.
- Japan Plant Protection Association, Ed.; *Guide for Use of Pheromone Formulations*; Japan Plant Protection Association: Tokyo, 2000; 112 pp (In Japanese).
- Honnda, Y.; Matsumoto, Y. Chapter 3-2: attractant originated from plants. In *Use Manual of Biological Pesticide and Growth Regulator*; Okada, S., Saka, S., Tamaki, Y., Motoyosi, H., Eds.; ELC: Tokyo, 1987; 214–233 (In Japanese).
- Sugie, H.; Tamaki, Y. Mass-trapping. Chapter 3-1: pheromone. In *Use Manual of Biological Pesticide and Growth Regulator*; Okada, S., Saka, S., Tamaki, Y., Motoyosi, H., Eds.; ELC: Tokyo, 1987; 181–183 (In Japanese).
- Wakamura, S. Practical use of synthetic sex pheromone for pest management. *Shyokubutsu-Boueki* **1993**, *47*, 499–503 (In Japanese).

Mating Disruption

Ring Carde

Department of Entomology, University of California, Riverside, Riverside, California, U.S.A.

INTRODUCTION

Mating disruption is a technique that prevents mating of a target pest by treating the crop to be protected with a formulated, synthetic copy of the insect's attractant sex pheromone. The omnipresence of the synthetic pheromone (termed a disruptant) interrupts the insect's ability to locate natural pheromone emitters and thereby reduces or eliminates mating.

MATING DISRUPTION

Mate location in moths generally is mediated by pheromones. Usually the female emits the chemical message while perched, and the male locates the female by flying upwind along her odor plume, sometimes over distances of tens of meters. The pheromone may have one to as many as six chemical components. The message is usually species specific and such specificity is typically achieved by use of unique blends and ratios of components. The majority of moth pheromones are 12-, 14-, or 16-carbon chain compounds, with one or two double bonds along the chain, and a terminal acetate, alcohol, or aldehyde moiety. Once pheromones of major moth pests were identified in the 1970s, it became possible to evaluate whether application of formulated synthetic pheromone onto crops disrupted normal orientation sufficiently to prevent mating and thereby achieve crop protection. In general, moth pheromones are not hazardous and they degrade quickly after application. The application rates are very low compared to conventional insecticides, usually on the order of 10 g/ha/week. Because of the high specificity of the pheromone message, a given formulation can be expected to disrupt mating only of the target pest. This is an advantage in that it avoids detrimental effects on beneficial arthropods, and therefore mating disruption can enhance biological control of secondary pests. However, the cost of control of the target species must justify the cost of applying mating disruptant treatments.

There are many cases of highly successful management of pest moths using this approach. The pink bollworm (*Pectinophora gossypiella*) (Gelechiidae) is a major pest of cotton in most cotton-producing regions of the world. Because its larvae feed internally in flower

buds and cotton bolls, it is a difficult pest to control, and it has developed resistance to some insecticides. In any case, such sprays can trigger outbreaks of secondary pests. The female's pheromone is a 1:1 mix of (Z,Z)-7,11- and (Z,E)-7,11-hexadecadienyl acetates. This mixture has been formulated in hollow plastic fibers, closed polyethylene tubes, plastic laminate flakes, and microcapsules. Most of these formulations are applied aerially at rates of approximately 10 g/ha and last 1 or 2 weeks. The plastic tube formulation is hand applied to the base of the cotton plant at 80 g/ha, and it lasts throughout the growing season. Successful commercial control of pink bollworm has been demonstrated in several regions (e.g., in Egypt on 50,000 ha in 1993). In a multiyear, area-wide management program of pheromone application on 11,000 ha in Parker, Arizona, the percentage of cotton boll damage by pink bollworm larvae declined precipitously from a preprogram level of 23% in 1989 to 0% in 1993.

The oriental fruit moth [*Grapholita* (= *Cydia*) *molesta*] (Tortricidae) is a major pest of peaches and nectarines throughout the stone-fruit growing areas of the world. This pest is difficult to control with broad-spectrum insecticides and some populations are insecticide resistant. The female's pheromone is a blend of (Z)-8- and (E)-8-dodecenyl acetates (in a ratio of 95:5) and (Z)-8-dodecen-1-ol (at 3–10% of the acetates). In trials in California and Virginia, a closed plastic tube formulation was placed in the upper third of fruit trees at 1000 dispensers/ha (75 g pheromone/ha) in two applications; the first was at the initiation of moth flight in the spring and the second was 90 days later. Control of this pest was at least as efficacious as with conventional insecticide treatments. A spectacular example of direct control of the oriental fruit moth was demonstrated in 1200 ha of peaches and nectarines in South Africa in 1991–1992. Although some orchards were heavily infested in the previous season, not a single infested fruit was found in orchards treated with the closed plastic tube formulation. These and other trials show that mating disruption is equivalent or superior to conventional insecticide treatment.

The tomato pinworm (*Keiferia lycopersicella*) (Gelechiidae) is a pest of tomatoes grown in Mexico, southern California, southern Texas, and Florida. Its economic damage stems mainly from larval entry into the fruit and this pest is resistant to many insecticides.

In Mexico, very high levels of fruit damage can occur, even with 20–45 applications of insecticide “cock-tails”/crop. The female’s pheromone is comprised of a single component, (*E*)-4-tridecyl acetate. Hollow fiber formulation applied by hand at 1000 release sites/ha with 10 g of pheromone provides complete protection of the tomatoes from pinworm damage and facilitates an integrated approach relying on parasite (*Trichogramma*) release, *Bacillus thuringiensis* and avermectin for management of 3 noctuid moth pests.

In Australia and New Zealand, control of the light-brown apple moth (*Epiphyas postvittana*) (Tortricidae) necessitates 10–12 insecticide applications/season. However, the recent evolution of organophosphate resistance in New Zealand means that this schedule often does not provide sufficient control for export apples. The pheromone is a 20:1 mixture of (*E*)-11-tetradecyl and (*E,E*)-9,11-tetradecadienyl acetates. In this case, control comparable to a conventional program of insecticide sprays has been achieved by use of a reduced spray schedule (6–7 sprays) combined with 1000 disruptant dispensers/ha.

Although the codling moth [*Cydia* (= *Laspeyresia*) *pomenella*] (Tortricidae) is a key pest of apple, it also infests pear, peach, apricot, and walnut. The major component of its pheromone is (*E,E*)-8,10-dodecadien-1-ol. Direct control of this pest by mating disruption in small orchards surrounded by conventional practice orchards has been quite inconsistent, very likely due to the influx of mated codling moth females from outside the treatment areas. Area-wide management programs, however, which typically encompass areas of several hundred or more hectares, have provided equivalent or improved control over conventional insecticide regimes. These programs require population levels to be low initially (below 1% fruit infestation or early season application of insecticide to reach these levels), and requires a vigorous monitoring program using pheromone-baited traps. Such integrated management programs are in wide use in the apple-growing regions of the Pacific Northwest of North America. In 2000, mating disruption of codling moth was used in Washington on 45% of pome fruits on over 40,000 ha. Impacts of mating disruption have

been less fruit damage and a more than 75% reduction in the use of broad spectrum insecticides, especially organophosphates.

These examples demonstrate that mating disruption can achieve direct control of some important moth pests, but they also illustrate that the mating disruption technique must be integrated into an overall management program. A keystone of all such programs is effective monitoring of pest density, so that if “remedial” application of conventional insecticides is required, as could be the case with the codling moth, these are applied before or with pheromone treatment. Some moth pests, such as the oriental fruit moth and the tomato pinworm, however, seem vulnerable to mating disruption even if populations are at high initial densities. In the case of the codling moth, the larger the treatment area and the more remote it is from the immigration of mated females, the more effective mating disruption is in crop protection.

Not all moth pests are likely to prove susceptible to mating disruption. If the moth is migratory, then the immigration of mated females into the area to be protected may result in unacceptable crop losses. Many noctuid pests (e.g., moths in the genera *Heliothis* and *Helicoverpa*) fly long distances and infest many crops. Control of such pests by mating disruption might only be feasible in area-wide management programs. Many insect groups besides moths use pheromones in mate location and aggregation, sometimes in conjunction with plant host volatiles. To date, the mating disruption approach has not proven efficacious with these nonmoth pests.

BIBLIOGRAPHY

- Cardé, R.T.; Minks, A.K. Control of moth pests by mating disruption: successes and constraints. *Annu. Rev. Entomol.* **1995**, *40*, 559–585.
- Cardé, R.T.; Minks, A.K. *Insect Pheromone Research: New Directions*; Chapman & Hall: New York, 1996; 684 pp.
- Howse, P.; Stevens, I.; Jones, O. *Insect Pheromones and Their Use in Pest Management*; Chapman & Hall: London, 1998; 369 pp.

Mechanical Weed Control in Agriculture

Charles L. Mohler

Department of Crop and Soil Sciences, College of Agriculture and Life Sciences,
Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

Mechanical weed control is the use of machinery or hand tools to physically damage weeds. It is the oldest form of weed management, and despite increased use of herbicides in recent decades, it is still a critical component of weed control programs in most cropping systems throughout the world. Mechanical weed control occurs during three time intervals in the cropping cycle: 1) during tillage prior to planting; 2) after planting by use of cultivation equipment specifically designed to remove weeds from the crop; and 3) during fallow seasons. Objectives of mechanical control during tillage include chopping and burying above-ground shoots, damaging below-ground perennating organs, and, in some cases, burying weed seeds too deeply for successful emergence. Cultivation after planting is aimed primarily at destroying recently emerged annuals, and reducing the vigor of perennials. During fallow periods, mechanical control methods can be used to prevent seed production, reduce density of seeds and buds in the soil, and prevent depletion of soil water prior to the next crop. Regardless of the timing of the operation, important considerations include the depth of soil disturbance and degree of soil inversion. Cultivation tools designed for use after crop planting further differ in the position they work relative to the crop row. *Full-field* cultivators work without regard to the row; *inter-row* cultivators work only the spaces between crop rows; *near row* tools work within a few centimeters of the row; and *in-row* tools work a band directly over the row.^[1] Information on different types of implements is summarized in Table 1. Post-planting cultivation has been made easier by recent advances in cultivator guidance.

TILLAGE PRIOR TO PLANTING

Tillage is most effective for weed management if 1) a large proportion of annual weeds have already emerged^[29] and 2) the buds of perennials have sprouted but the shoots have not yet replenished carbohydrates in storage organs.^[30] Recently developed models use soil temperature and moisture to predict the percentage of total weed emergence for many widespread weed species.^[29,31]

Tillage buries weed seeds that were shed since the last tillage event, and brings buried seeds to the surface where they may be stimulated to germinate by light, improved gas exchange, warmer soil, and diurnal fluctuations in soil temperature.^[32] Whether tillage increases or decreases density of an annual thus depends on a complex but explicable interaction between the seed rain in previous years, types of tillage employed in past years and in the current year, the survival of the weed species as a function of depth in the soil, and the species' emergence in response to depth.^[33] In general, species with short longevity in the seed bank are most easily managed with deep inversion tillage because many seeds die before returning to the surface. In contrast, species with potentially great longevity are often best managed with no or minimal tillage because seed mortality is usually greater near the soil surface. Tillage at night or with light-shielded implements typically reduces emergence of light sensitive weeds by 20% to 50%.^[34,35]

CULTIVATION AFTER CROP PLANTING

Full-field implements work very shallowly, usually no deeper than the planting depth of the crop. They are thus effective only against small seeded weeds that lack sufficient reserves for emergence from deep in the soil. Fortunately, however, this includes most agricultural weed species. The principal types of full-field implements are weeding harrows and rotary hoes. Weeding harrows consist of many downward pointing small-diameter tines that drag through the soil, breaking and burying small weeds.^[20] Rotary hoes consist of tiers of closely ranked, ground-driven wheels with spoon-like spokes that flick soil and small weeds into the air.^[19] Weeding harrows and rotary hoes are most effective against weeds in the white thread and early cotyledon stages.^[17] They work best when the soil has recently been wet enough for germination, but is sufficiently dry to be crumbled by the implement.^[17] These tools are typically used pre-emergence and once or twice post-emergence up to a crop height of about 15 cm.^[18–20,22]

Inter-row cultivators generally carry either shank-mounted sweeps or shovels, or else ground or power-take-off-driven rotating tines. In any case, these are

robust machines that thoroughly cut, dig, and bury even relatively large weeds. They generally cannot work close to the crop due to danger of root pruning.^[36] Shields reduce the risk of burying the crop when it is small. The most common types of inter-row cultivators have three or five shovels per inter-row. S-shaped shanks are popular on these machines because the vibrating action of the shank shakes soil from weed roots.^[37] Implements designed for use in reduced tillage systems generally have a single wide sweep per inter-row, a coulter in front of the shank to cut crop residue, and high clearance to allow crop residue to flow freely through the machine.^[2] Most have heavy C-shaped shanks to provide stability in unbroken soil.

Ground-driven rolling cultivators and power-driven inter-row rotary tillers are used primarily in vegetable production. Rolling cultivators carry gangs of wheels with curved tines. They are less aggressive against large weeds than shovel cultivators, but they are highly flexible implements that can, for example, cultivate the sides of raised beds or dig more shallowly close to the row than in the row middle. Rotary tillers are very aggressive and create a loose soil surface that inhibits weed germination, but they can cause loss of soil structure due to excessive pulverization. Inter-row cultivators of all types are often configured to bury small weeds in the row with soil once the crop becomes large.^[3,38]

Near-row tools include L-shaped vegetable knives, disk hillers, spyders, basket weeders, and brush weeders. The first three types mount on inter-row cultivators whereas the latter two cultivate the whole inter-row area. Vegetable knives are low-pitched half-sweeps. They usually point away from the row when the crop is small, but can be reversed to cultivate under the crop canopy when the crop grows larger.^[39] Disk hillers are sharp wheels that cut and dig out weeds close to the row. Spyders have curved, spoked wheels that dig out the weeds. Both disk hillers and spyders are usually mounted to move soil away from the row when the crop is young but can be reversed to throw soil into the row later in the season.^[7,9] Basket weeders consist of pairs of counter-rotating wire baskets. Horizontal axis brush weeders have power-driven rotating plastic brushes. Both have the axis of rotation at right angles to the crop row, which causes most soil movement to be parallel to the row. This, plus shields on the brush weeder, allows cultivation within a few centimeters of the row.^[40] Both basket and brush weeders thoroughly sweep out and maul all small weeds and leave a loose soil surface that inhibits weed germination. Brush weeders are one of the few implements that can be used in wet soil.^[13]

In-row tools include torsion weeders, spring hoes, spinners, rubber finger weeders, vertical axis brush weeders, electrocution weeders, and in-row flame

weeders. Torsion weeders and spring hoes work by compressing the surface soil in the crop row between spring steel wires or sheets, respectively. This causes the soil to boil up, thereby damaging seedlings in the white thread or cotyledon stages.^[9] Spinners are ground-driven, open-ended spring steel baskets. The tines scratch laterally across the row, thereby uprooting small weeds.^[8] All three implements mount on conventional inter-row cultivators. Field tests have shown improved weed control with these implements relative to shovel cultivation alone.^[3,8,41] Rubber finger weeders work the in-row line with wheels of rubber fingers that flex around firm crop stems but uproot small weed seedlings.^[16] Vertical axis brush weeders brush out small weeds around and between slightly larger or better rooted crop plants.^[14] All in-row implements require careful depth control and positioning relative to the row to avoid crop damage. Because they work best against very small weeds in a well-established crop, usually early flushes of weeds will be removed with a full-field implement (e.g., Ref.^[3]). In-row flame weeders kill small weeds by disrupting plant tissues with a propane flame. Their use is restricted to crops like maize and onions that have a protected bud, and cotton, which has a corky stem.^[42,43]

Cultivator guidance systems reduce operator fatigue and crop damage, and potentially increase the speed of cultivation. Mechanical systems use disks or cones to guide off furrows made by the planter or the sides of raised beds.^[3,44] They are inexpensive and allow guided cultivation even when the crop is small, but they require that the bed or furrow be maintained through multiple operations. Electronic guidance systems sense the crop with feelers and then mechanically reposition the cultivator.^[44,45] These systems are effective only when the crop is large enough to sense. Systems that work from video images are under development^[46] and may allow guided cultivation of smaller crops.

MECHANICAL WEED MANAGEMENT DURING FALLOW PERIODS

Reducing seed production is an important component of integrated weed management. Mowing or tillage after crop harvest can often greatly reduce seed production by weeds that have grown up within the crop but are not yet mature.^[47]

Repeated shallow tillage interspersed with rests of one to a few weeks is often effective for flushing germinable seeds from the seed bank.^[48,49] This “false seedbed” technique can be applied either after crop harvest or prior to planting, depending on the germination ecology of the weed species present in the seed bank.

Repeated tillage during fallow seasons can also be used to weaken perennial species by forcing the release

Table 1 Operating parameters, uses, and limitations of various types of mechanical weeding tools and implements

| Implement/tool ^a | Position of action | Operating depth ^b (cm) | Speed ^c (km hr ⁻¹) | Weed size ^d (cm) |
|-------------------------------------|-------------------------------|--------------------------------------|--|---|
| Shovels and sweeps (hoes) | Inter-row | (3) 5 to 8 (10) | 2 to 8 (10) | To large size |
| Rolling cultivator— spyder gangs | Inter-row, near row | (2) 5 to 7 | 2 to 8 | To 30+ |
| Rolling cultivator— disk gangs | Inter-row | (2) 5 to 7 | 2 to 8 | To large size |
| Horizontal disk cultivator | Inter-row | 2 to 6 | 6 to 13 | To 40 |
| Rotary tiller (power hoe) | Inter-row | 3 to 8 | 2 to 8 | To large size |
| Mower | Inter-row | None | 4 to 10 | To large size |
| Disk hillers (cutaway disks) | Near row | 2 to 7 | 2 to 8 | To large size |
| Spyders | Near row | 2 to 5 (7) | 2 to 8 | To 30+ |
| Basket weeder | Very near row to inter-row | 2 to 3 | 6 to 10 | To 3 |
| Brush weeder— horizontal axis | Very near row to inter-row | 3 to 5 | 2 to 5 | Uproots seedlings, strips larger weeds |
| Brush weeder— vertical axis | Near row, in row | 1 to 4 | 0.5 to 4 | To ~10 |
| Torsion weeders, spring hoes | In row | 2 to 3 | 2 to 8 | Thread to cotyledon |
| Spinners | In row | 2 to 5 | 2 to 8 | Thread to cotyledon (5) |
| Rubber finger weeder | In row | 2 | 2 to 8 | Thread to cotyledon |
| Rotary hoe | Full-field | 2 to 4 | 11 to 21 | Thread to cotyledon |
| Spring-tine harrow | Full-field | 2 to 5 (10) | 3 to 8 (12) | Thread to cotyledon |
| Spike harrow, chain harrow | Full-field | 2 to 5 | 3 to 8 (12) | Thread to cotyledon |
| Rod weeder | Full-field | 4 to 6 | 8 to 12 | To large size |
| Flame weeder | Full field or in row | None | 1 to 7 (13) | Cotyledon to 5 (20)cm |
| Hot water weeder | Inter-row, full field | None | 2 to 6 | To large size |
| Electrocution weeder | Full field | None | 2 to 5 | 10 to 20 cm taller than crop |
| Weed puller | In row | None | ~5 | 10 to 15 cm taller than crop |
| Rotary orchard weeder | In row and near row | 2 | 8 to 11 | To large size |

^aImplements are in singular form, tools that attach to another implement are given in plural. Synonyms are given in parentheses.

^bUnusual operating depths that are used in some circumstances are given in parentheses.

^cUnusual operating speeds that are used in some circumstances are given in parentheses.

^d“To large size” indicates that the implement is effective against even large weeds. The upper size limit will vary with weed species and operating conditions, but is generally not a limitation for the implement.

^e“Limit set by clearance” indicates that the implement can be used until the crop has spread laterally so much that it is crushed by tractor tires, or is so tall that it will no longer pass under the tractor axle or implement tool bar.

^fMuch information in this table is based on the author’s personal experience, materials supplied by manufacturers, and discussions with growers and other researchers.

^gThe implement is effective in most row crops, but is largely limited to high-value crops because of the need for time consuming adjustments, flat seed bed, slow operating speed, etc.

| Crop size ^e (cm) | Soil movement | Crops | Soil limitations | References ^f |
|---------------------------------|----------------------|--|---|-------------------------|
| Limit set by clearance | Toward row | All row crops | Few soil limitations High residue models available | [2] |
| Limit set by clearance | Directional choice | All row crops, sides of beds | Tolerates moderate rockiness | [3–5] |
| Limit set by clearance | Directional choice | All row crops | Poor in residue Tolerates moderate rockiness and high residue | [5] |
| Limit set by clearance | Toward row | Most row crops | Tolerates moderate residue | [5] |
| Limit set by clearance | Random | Most row crops ^g | Reduces soil structure | [6] |
| Limit set by clearance | None | Most row crops | Problem with surface rocks | |
| Limit set by clearance | Directional choice | All row crops | Few limitations | [7] |
| Limit set by clearance | Directional choice | All row crops | High residue model available | [3,8,9] |
| 2 to 25 | Parallel to row | Most row crops, ^g tree seedlings | Intolerant of rocks. Best with flat seedbed | [10] |
| To 20 (28) | Parallel to row | Most row crops, ^g tree seedlings, (cereal) | Tolerates wet soil | [11–13] |
| Limit set by clearance | Directional choice | Many row crops | Rocks may jam shields Best with flat seedbed Tolerates wet soil | [14,15] |
| Limit set by clearance | Slight toward row | Most row crops, ^g heavy model for tree and vine crops | Tolerate minor rockiness and residue | [3,8,9] |
| To 10 (20) | Minimal | Many row crops ^g | Tolerate moderate rockiness | |
| 25 (40) | Minimal, or from row | High value row crops, nursery stock | Poor in crusted soil, large residue | [16] |
| To 15 | Random | Large seeded crops, cereals | Tolerates moderate rockiness Poor in wet soil | [3,17–19] |
| To 15 (20) | Random | Large seeded crops, cereals, transplants | High residue models available Poor in residue, crusted soil | [20–23] |
| To 15 | Random | Large seeded crops, cereals | Poor in residue | [20,22] |
| Fallow, post-harvest | Minimal | Primarily dryland fallow | Tolerates residue | [24,25] |
| Mostly pre-emergence | None | Pre-emergence in most crops | Fire hazard in dry residue | [26,27] |
| To large size in a few crops | | Post-emergence in crops with protected buds | | |
| Tall woody crops | None | Tree and vine crops | Few soil limitations | |
| To 80 (100) | None | Low growing row crops | Best with dry soil | [28] |
| To 80 (100) | Minimal | Low growing row crops | Tolerates rocks and residue | |
| Tall woody | From row | Tree and vine crops | Best in wet soil Tolerates rocks and residue | |

of dormant buds and use of stored carbohydrates.^[30,50,51] Tillage during fallow periods can also be used to expose perennating organs to desiccation^[52] or cold damage.^[53] Perennating organs can sometimes be worked to the soil surface and removed by raking.

CONCLUSIONS

New machinery and the application of old machinery with new insights allows continuing improvement in mechanical weed control. This ancient approach to weed management continues to play a critical role in agriculture, even within the most modern integrated weed management systems.

[See also Flame Weeding in Corn, Weed Electrocutation, Soil Cultivation, Fallows, and Tillage.]

REFERENCES

- Mohler, C.L. Mechanical management of weeds. In *Ecological Management of Agricultural Weeds*; Liebman, M., Mohler, C.L., Staver, C.P., Eds.; Cambridge University Press: New York, 2001; 139–209.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 18–21.
- Mohler, C.L.; Frisch, J.C.; Mt. Pleasant, J. Evaluation of mechanical weed management programs for corn (*Zea mays*). *Weed Technol.* **1997**, *11*, 123–131.
- Mt. Pleasant, J.; Burt, R.F.; Frisch, J.C. Integrating mechanical and chemical weed management in corn (*Zea mays*). *Weed Technol.* **1994**, *8*, 217–223.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 23 pp.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 64–65.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 26–37.
- Schweizer, E.E.; Westra, P.; Lybecker, D.W. Controlling weeds in corn (*Zea mays*) rows with an in-row cultivator versus decisions made by a computer model. *Weed Sci.* **1994**, *42*, 593–600.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 58 pp.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 60 pp.
- Lampkin, N. *Organic Farming*; Farming Press: Ipswich, UK, 1990; 196–199.
- Pedersen, B.T. Test of the multiple row brush hoe. *Veröff. Bundesanst. Agrarbiol. Linz/Donau* **1990**, *20*, 109–125.
- Weber, H. Mechanical weed control with a row brush hoe. *Acta Hort.* **1994**, *372*, 253–260.
- Fogelberg, F.; Dock Gustavsson, A.-M. Mechanical damage to annual weeds and carrots by in-row brush weeding. *Weed Res.* **1999**, *39*, 469–479.
- Melander, B. Optimization of the adjustment of a vertical axis rotary brush weeder for intra-row weed control in row crops. *J. Agric. Eng. Res.* **1997**, *68*, 39–50.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 59 pp.
- Lovely, W.G.; Weber, C.R.; Staniforth, D.W. Effectiveness of the rotary hoe for weed control in soybeans. *Agron. J.* **1958**, *50*, 621–625.
- Mulder, T.A.; Doll, J.D. Integrating reduced herbicide use with mechanical weeding in corn (*Zea mays*). *Weed Technol.* **1993**, *7*, 382–389.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 14–15.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 16–17.
- Peruzzi, A.; Silvestri, N.; Gini, N.; Coli, A. Weed control of winter cereals by means of weeding harrows: first experimental results. *Agric. Mediterr.* **1993**, *123*, 236–242.
- Rasmussen, J.; Svenningsen, F. Selective weed harrowing in cereals. *Biol. Agric. Hortic.* **1995**, *12*, 29–46.
- Rydberg, T. Weed harrowing—The influence of driving speed and driving direction on degree of soil covering and the growth of weed and crop plants. *Biol. Agric. Hortic.* **1994**, *10*, 197–205.
- Anonymous. Rod weeder... What it does, how it works. *World Farming* **1978**, *20*, 58–59.
- Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 92 pp.
- Ascard, J. *Thermal Weed Control by Flaming: Biological and Technical Aspects*; Report 200; Department of Agricultural Engineering, Swedish University of Agricultural Sciences: Alnarp, 1995; 1–61.
- Ascard, J. Effects of flame weeding on weed species at different developmental stages. *Weed Res.* **1995**, *35*, 397–411.
- Vigneault, C.; Benoit, D.L.; McLaughlin, N.B. Energy aspects of weed electrocutation. *Rev. Weed Sci.* **1990**, *5*, 15–26.
- Forcella, F. Real-time assessment of seed dormancy and seedling growth for weed management. *Seed Sci. Res.* **1998**, *8*, 201–209.
- Håkansson, S. Multiplication, growth and persistence of perennial weeds. In *Biology and Ecology of Weeds*; Holzner, W., Numata, N., Eds.; Dr. W. Junk Publishers: The Hague, 1982; 123–135.
- Reese, C.; Forcella, F. *Weed Cast User's Manual in MS Word 2.0 for Windows Format*; <http://www.infolink.morris.mn.us/~1wink/products/weedcast.htm> (accessed August 2000).
- Fenner, M. Ecology of seed banks. In *Seed Development and Germination*; Kigel, J., Galili, G., Eds.; Marcel Dekker: New York, 1995; 507–528.

33. Mohler, C.L. A model of the effects of tillage on emergence of weed seedlings. *Ecol. Appl.* **1993**, *3*, 53–73.
34. Buhler, D.D. Effects of tillage and light environment on emergence of 13 annual weeds. *Weed Technol.* **1997**, *11*, 496–501.
35. Scopel, A.L.; Ballaré, C.L.; Radosevich, S.R. Photostimulation of seed germination during soil tillage. *New Phytol.* **1994**, *126*, 145–152.
36. Russel, W.J.; Fehr, W.R.; Mitchell, R.L. Effects of row cultivation on growth and yield of soybeans. *Agron. J.* **1971**, *63*, 772–774.
37. Buckingham, F.; Pauli, A.W. *Tillage*, 3rd Ed.; Deere & Company: Moline, Illinois, 1993; 112–113.
38. Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 37–38, 47.
39. Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 40, 79–80.
40. Ascard, J.; Mattson, B. Inter-row cultivation in weed-free carrots: the effect on yield of hoeing and brush weeding. *Biol. Agric. Hortic.* **1994**, *10*, 161–173.
41. VanGessel, M.J.; Schweizer, E.E.; Wilson, R.G.; Wiles, L.J.; Westra, P. Impact of timing and frequency of in-row cultivation for weed control in dry bean (*Phaseolus vulgaris*). *Weed Technol.* **1998**, *12*, 548–553.
42. Ascard, J. Thermal weed control with flaming in onions. *Veröff. Bundesanst. Agrarbiol. Linz/Donau* **1990**, *20*, 175–188.
43. Seifert, S.; Snipes, C.E. Influence of flame cultivation on mortality of cotton (*Gossypium hirsutum*) pests and beneficial insects. *Weed Technol.* **1996**, *10*, 544–549.
44. Bowman, G., Ed.; *Steel in the Field: A Farmer's Guide to Weed Management Tools*; Sustainable Agriculture Network: Beltsville, Maryland, 1997; 30–33.
45. Bowman, G. Killer cultivators: guidance systems worthy of NASA make weeding faster, easier. *New Farm* **1991**, *13* (3), 16–35.
46. Tian, L.; Slaughter, D.C.; Norris, R.F. Outdoor field machine vision identification of tomato seedlings for automated weed control. *Trans. Am. Soc. Agric. Eng.* **1997**, *40*, 1761–1768.
47. Kegode, G.O.; Forcella, F.; Durgan, B.R. Limiting green and yellow foxtail (*Setaria viridis* and *S. glauca*) seed production following spring wheat (*Triticum aestivum*) harvest. *Weed Technol.* **1999**, *13*, 43–47.
48. Darwent, A.L.; Smith, J.H. Delayed seeding for wild oat control in rapeseed in Northwest Alberta. *Can. J. Plant Sci.* **1985**, *65*, 1101–1106.
49. Johnson, W.C., III; Mullinix, B.G., Jr. Stale seedbed weed control in cucumber. *Weed Sci.* **1998**, *46*, 698–702.
50. Bourdôt, G.W.; Butler, J.H.B. Control of *Achillea millefolium* L. (yarrow) by rotary cultivation and glyphosate. *Weed Res.* **1985**, *25*, 251–258.
51. Willard, T.R.; Shilling, D.G.; Gaffney, J.F.; Currey, W.L. Mechanical and chemical control of cogon-grass (*Imperata cylindrica*). *Weed Technol.* **1996**, *10*, 722–726.
52. Phillips, M.C. Use of tillage to control *Cynodon dactylon* under small-scale farming conditions. *Crop Prot.* **1993**, *12*, 267–272.
53. Schimming, W.K.; Messersmith, C.G. Freezing resistance of overwintering buds of four perennial weeds. *Weed Sci.* **1988**, *36*, 568–573.

Mechanisms of Resistance to Agrochemicals

Derek Hollomon

Department of Agricultural Sciences, Long Ashton Research Station, Institute of Arable Crops Research, University of Bristol, Bristol, U.K.

INTRODUCTION

Living organisms are intrinsically variable, and exposure to pesticides selects individuals best able to survive. In the laboratory, selection can be intensified using mutagenic agents such as UV light and *N*-nitrosoguanidine to increase the frequency of mutation, and resistance has been generated in at least 1000 insects, 500 plant pathogens, and 100 weeds through laboratory studies. But this overstates the problem of resistance in practice because, although similar mutations can be found in field populations, the majority carry a fitness penalty and do not compete well in the absence of pesticides. Furthermore, organisms favored for laboratory studies are often of limited economic importance, and so the number of species encountering serious resistance, and consequently control difficulties, is probably no more than 50. Where resistance emerges rapidly, it often reflects a target site change and a mutation in a single major gene; where it evolves gradually, several mechanisms and genes (polygenes) may be involved. Over time, major gene and polygenic resistance mechanisms may combine generating complex cross-resistance patterns, and serious difficulties developing effective antiresistance strategies. Resistance is not confined to a particular species, but in general pests, diseases, and weeds with short generation times and high reproduction rates, all have a high risk of developing resistance.

TARGET SITE CHANGES

Mutations that alter the binding site of a pesticide to its target can generate high levels of resistance. The mutation must carry little or no fitness penalty and must allow the target protein to function normally. Generally, these are point mutations altering only a single amino acid. Despite the fact that many different point mutations conferring resistance can be generated in a target protein in the laboratory, most are never recovered from field populations. Instead resistance is usually confined to one or two tightly defined regions of the protein and, surprisingly, in highly conserved regions where one might expect function to be impaired. Key examples of target site changes associated with

high levels of resistance and performance difficulties involve some herbicides that inhibit acetyl coenzyme A carboxylase (ACCase) or ALA synthetase, insecticides that interact with the sodium channel or acetylcholinesterase interfering with nerve function, or benzimidazole, DMI and strobilurin (QOI) fungicides. The same point mutation confers resistance across species, genera (Table 1) and even wider boundaries in the case of benzimidazoles used in veterinary medicines against parasitic worms.

Other changes at the target site can contribute to resistance. Overexpression leads to more target protein, and consequently more pesticide is needed to inhibit it. This may simply result from more efficient transcription and translation. Target site proteins, such as beta-tubulin and cytochrome *P450s*, belong to gene families. Many fungi contain more than one beta-tubulin protein, and each may function at a different stage of development. When one member of a gene family is inactivated by a pesticide, another may function in its place, but how important this mechanism is outside laboratory-resistant mutants is not clear.

METABOLISM

Pesticides cross many boundaries before reaching their targets. If systemic, they enter and are mobile within the plant. Insecticides and fungicides must go further and enter insects and fungi, respectively. Selective toxicity is a delicate balance between key features of the chemistry, and detoxification by a few enzymes whose action makes pesticides more water-soluble. This is especially linked to excretory systems in insects, and the ability of plants to partition unwanted metabolites into vacuoles. Oxidation by mixed-function oxidases (cytochrome *P450s*) contributes in black grass (*Aleporurus myosuroides*) to detoxification of many herbicides, while conjugation with glutathione offers another mechanism to increase polarity, and generate resistance. Nonspecific esterases produce free acids, and this often destroys pesticide activity. In fungi, this is a less attractive option for resistance since they lack much of the enzyme machinery needed to generate water-soluble products. A common feature of metabolic resistance is that cross-resistance generally extends to

Table 1 A single point mutation in the mitochondrial cytochrome *bcl* at amino acid codon 143 (G 143 A) gene is linked to strobilurin resistance

| Pathogen | Disease | Phenotype | Amino acid sequence | |
|---|-------------------------|-----------|---------------------|-----|
| | | | 135 | 152 |
| <i>Venturia inaequalis</i> | Apple scab | Wild-type | PYGMSLWGATVITNLMS | |
| | | Resistant | PYGMSLWAATVITNLMS | |
| <i>Erysiphe graminis</i> f.sp. <i>tritici</i> | Wheat powdery mildew | Wild-type | PYGMSHWGATVITNLMS | |
| | | Resistant | PYGMSLWAATVITNLMS | |
| <i>Sphaerotheca fuliginea</i> | Cucurbit powdery mildew | Wild-type | PYGGMSFWGAT | |
| | | Resistant | PYGGMSFWAAT | |
| <i>Mycosphaerella fijiensis</i> | Banana Sigatoka | Wild-type | PYGMSLWGATVITNLMS | |
| | | Resistant | PYGMSLWAATVITNLMS | |

(Data from W. Koeller (*V. inaequalis*), H. Ishii (*S. fuliginea*), and B. Fraaije (*E. graminis* and *M. fijiensis*), personal communications, 2000.)

pesticides with many different modes of action, whereas in fungi, cross-resistance is generally confined to fungicides with the same mode of action.

In the case of organophosphorous or carbamate insecticides, resistance in certain aphids (e.g., *Myzus persicae*), results from amplification of an esterase gene. Increased amounts of esterase sequester these insecticides and cause resistance. Changes in the extent of methylation in the upstream promoter region of an esterase gene govern the level of amplification. One practical feature of this resistance is that it is readily reversed in the absence of selection, although the outcome of this maneuver depends on whether the promoter changes and mutations are lost, or simply that transcription or translation is reduced. If the latter, aphids rapidly regain their resistance when selection is imposed again.

MULTIDRUG RESISTANCE

Fungi may lack possibilities for resistance through metabolic detoxification, but have instead membrane-bound transporter proteins which enable the efflux of unwanted molecules, including fungicides, into the surrounding medium. Energy is needed to move lipophilic fungicides against a concentration gradient, and so transporter proteins possess ATPase driven pumps. Their activity can be increased through over-expression, which is often induced by chemically unrelated molecules. Consequently, this nonspecific process, which is termed multidrug resistance (MDR), can lead to resistance through increased efflux of fungicides, a process that can be blocked by inhibitors of ATP synthesis. Although MDR may account for clinical resistance to fluconazole and related azole drugs, in plant pathogens this mechanism has yet to be linked to practical resistance, although it may well augment problems associated with target site changes.

OTHER MECHANISMS

Many other resistance mechanisms have been identified in studies of resistance generated only in the laboratory. Alteration of metabolic pathways so as to avoid the target site; altering the pH of the surrounding medium affecting the degree of ionization and uptake of pesticides; and increasing the lipid content so that lipophilic molecules are partitioned away from more polar target sites have all been described as resistance mechanisms. Some pesticides require activation and this process can be blocked. Phosphorothiolate fungicides must undergo P-S and C-S cleavage before they inhibit phospholipid biosynthesis, and this can be blocked by inhibitors of mixed function oxidases, such as DMI fungicides. None of these mechanisms contribute significantly to resistance in practice on their own, but in combination with other mechanisms, they can cause important practical control problems.

IMPACT OF RESISTANCE MECHANISMS ON MANAGEMENT OF RESISTANCE

Metabolic detoxification and MDR are generally non-specific, and cross-resistance extends to pesticides with different modes of action. Antiresistance strategies involving mixtures with different modes of action are not viable options, although synergy acting through inhibitors of enzymes involved in detoxification is a powerful strategy where suitable synergists exist. Inhibitors of mixed function oxidases, such as piperonyl butoxide, are widely used to overcome resistance to certain insecticides. Target site resistance generates cross-resistance patterns confined to pesticides with the same mode of action, and this is a strong feature of fungicide resistance, but less so for insecticides and herbicides. This not only provides options for anti-resistance strategies involving mixture partners with

different modes of action, but it also creates possibilities for negative cross-resistance. Changes at the target site causing resistance to one group may allow better binding of another inhibitor, generating possibilities for antiresistance strategies based on these mixture partners. This has indeed been used in practice, exploiting the negative cross-resistance between benzimidazole and phenylcarbamate fungicides. Unfortunately, changes in the target beta-tubulin produced strains resistant to both fungicides, limiting the usefulness of this approach to maintaining the effectiveness of benzimidazole fungicides.

RAPID DIAGNOSIS OF RESISTANCE

Antiresistance strategies must be monitored to ensure that they remain effective. Bioassays are still the main component of resistance-monitoring exercises, and certainly where resistance mechanisms have not been identified, there are no other options. But bioassays are resource-intensive, especially for pests and diseases that grow slowly or are difficult to maintain. In some cases, rapid and cheap biochemical assays can act as useful monitoring tools, for example, in measuring esterase levels in certain insects. But where the molecular mechanisms of resistance are known, and the underlying DNA changes causing them identified, rapid diagnosis is an option available using powerful polymerase chain reaction (PCR) technologies. Point mutations, deletions, and inversions can all be detected through careful design of allele-specific probes and PCR primers, coupled to fluorescent markers. Harnessing recent developments surrounding "real-time PCR," "Taqman" chemistry, and molecular beacons allows detection of several resistance mutations in a single PCR assay, and at frequencies of 1:10,000 or lower. This level of detection is beyond the reach of bioassay methods. Where target-site resistance has been defined in one species, as in the case of strobilurin fungicides (Table 1), molecular diagnostic technologies offer opportunities for the early detection of the same mutation in other pests, diseases, and weeds.

FUTURE DIRECTIONS

Molecular biology has undoubtedly expanded understanding of resistance mechanisms. A practical outcome has already reached growers through herbicide-resistant crops. Molecular analysis of resistance also provides a springboard to exploit rapid diagnostic techniques, which improve the accuracy with which

changes in the frequency of resistance alleles can be followed in field populations of pests, disease, and weeds. The fitness of individual resistance mutations can now be evaluated under field conditions, rather than in limited populations in growth rooms where simulated environments seldom reflect natural conditions. Predictive modelling of the impact of different antiresistance strategies becomes a serious possibility, and this should help in the management of resistance. Key questions can be addressed in new ways. Why have some pesticides never developed resistance in practice, despite the generation of resistant mutants in laboratory studies? Coupled with recombinant DNA methods that provide large quantities of mutant and wild-type target proteins, and physical techniques to define structural changes, platforms can be established to search for new chemistry active against target sites with low resistance risk.

ACKNOWLEDGMENTS

The author is grateful for the many discussions with colleagues in the agrochemical industry and elsewhere that have contributed to ideas contained in this summary of resistance mechanisms. IACR Long Ashton is supported by a grant from the U.K. Biotechnology and Biological Sciences Research Council.

BIBLIOGRAPHY

- Brown, T.M., Ed.; *Molecular Genetics and Evolution of Pesticide Resistance*; American Chemical Society: Washington, DC, 1996.
- Denholm, I., Pickett, J.A., Devonshire, A.L., Eds.; *Insecticide Resistance: From Mechanisms to Management*; Proc. R. Soc. London, Series B., 1998; Vol.353 (1375).
- Hollomon, D.W.; Butters, J.A. Molecular determinants for resistance to crop protection chemicals. In *Molecular Biology in Crop Protection*; Marshall, G., Walters, D., Eds.; Chapman & Hall: London, 1994; 98–117.
- Joseph-Horne, T.; Hollomon, D.W. Molecular mechanisms of azole resistance in fungi. *FEMS Microbiol. Lett.* **1997**, 141–149.
- Lyr, H., Ed.; *Modern Selective Fungicides: Properties, Applications and Mechanisms of Action*, 2nd Ed.; Gustav Fischer: Jena, Germany, 1995.
- Powles, S.B., Holtum, J.A.M., Eds.; *Herbicide Resistance in Plants: Biochemistry and Biology*; Lewis Publishers: Boca Raton, FL, 1994.
- Roe, R.M., Burton, J.D., Kuhr, R.J., Eds.; *Herbicide Activity: Toxicology, Biochemistry and Molecular Biology*; IOS Press: Amsterdam, 1997.

Mitigating Impacts of Terrestrial Invasive Species

Kathleen Fagerstone

APHIS/WS, USDA - National Wildlife Research Center, Fort Collins, Colorado, U.S.A.

INTRODUCTION

Human beings have introduced other species around the world both accidentally and intentionally. Accidental introductions resulted from escape from captivity (monk parakeets [*Myiopsitta monachus*] in Florida), stowaways (rats [*Rattus* spp.] and house mice [*Mus musculus*] worldwide; brown treesnakes [*Boiga irregularis*] in Guam), or expansion of species' ranges. Intentional introductions occurred for various reasons including: 1) aesthetics (songbirds into Hawaii, grey squirrel [*Sciurus carolinensis*] into Europe, and European songbirds imported by British colonists into North America, Australia, and New Zealand); 2) economics (nutria [*Myocastor coypus*] introduced in the eastern U.S., and Arctic fox [*Alopex lagopus*] onto Aleutian Islands for development of fur industries); 3) recreation (pheasants [*Phasianus colchicus*] and chukar [*Alectoris chukar*] introduced as game species from Asia to North America, and red deer [*Cervus elaphus*] introduced into New Zealand); 4) food (domestic livestock worldwide, rabbits [*Oryctolagus cuniculus*] into Australia, pigs [*Sus scrofa*] into Hawaii); 5) for biological control (mongooses [*Herpestes auropunctatus*] to control rats in Hawaii, fox [*Vulpes vulpes*] to control rabbits in Australia, and giant toad [*Bufo marinus*] to control cane beetles in Australia); or 6) releases from captive populations (bulbuls [*Pycnonotus jocosus*] in Florida and domestic ferrets [*Mustela putorius*] in California, mink [*Mustela vison*] and muskrat [*Ondatra zibethicus*] in Europe, and horse [*Equus caballus*], donkey [*Equus asinus*], and other ungulates into Australia and western North America).

The majority of biological introductions fail. Of those that succeed, only a small fraction become serious pests. Many introductions, like livestock or pheasants into the U.S., have been generally beneficial; however, some introduced species become invasive, defined as non-native species which cause substantial economic or ecological harm. The U.S. has at least 221 non-native terrestrial vertebrate species^[1] and New Zealand has 35 introduced birds and 33 mammals, where previously the only mammals consisted of 3 bats.^[2] About 44 mammals have been introduced into Australia, of which 27 have become established,^[3] along with 3 species of amphibians and reptiles and numerous birds. Ten species of terrestrial mammals on the Galapagos are aliens.

CHARACTERISTICS OF INVASIVE SPECIES AND HABITATS VULNERABLE TO INVASION

Successful invading species tend to be native to extensive habitats within continents and can usually tolerate a wide variety of environmental conditions. Also, species in close association with humans (commensal), including rats, house mice, house sparrows (*Passer domesticus*), starlings (*Sturnus vulgaris*), and rock doves (*Columbia livia*), are most successful in invading other man-modified habitats.

Certain regions are most vulnerable to introduction of invasive species. For example, not all U.S. states are affected equally by invasive species. Particularly vulnerable are Hawaii and Florida, where a high percentage of terrestrial vertebrates are introduced. New Zealand, Australia, and Madagascar also have high percentages of introduced species. Several features account for the disproportionate number of introduced species in these areas.^[4,5] The primary feature is geographic isolation: Hawaii and New Zealand are island archipelagos, Florida is a peninsula bounded on three sides by water and on one side by frost zones, and Australia and Madagascar function as insular continents. A typical feature of islands and isolated areas is an impoverished native fauna relative to equal size mainland areas. Invasive species were successful on New Zealand and Hawaii because native species did not previously occupy similar niches. Australia is another example; because birds colonized across the water barrier, the native bird fauna is diverse and only two invasive bird species have spread into undisturbed habitats.^[6] In contrast, at least 12 species of mammals with no ecological counterparts in Australia have spread widely.^[3,6]

A mild climate also makes areas vulnerable to invasive species. Hawaii and Florida have large tropical or subtropical areas without freezing temperatures. The accidental escape of exotic pets like bulbuls or the introduction of tree frogs from nursery stock would be innocuous in most U.S. regions because of cold climates. In Florida and Hawaii they thrive and spread. Finally, locations vulnerable to introductions are transportation hubs. Most visitors from Latin America, and many from other regions, enter the U.S. through Miami, and Hawaii is a center for both civilian and military traffic moving throughout the Pacific.

ECONOMICAL AND ECOLOGICAL IMPACTS OF INVASIVE SPECIES

Pimentel et al.^[7] estimated that about 50,000 introduced species now inhabit the U.S. Not all have negative consequences, as they account for over 98% of the U.S. food system^[7] valued at \$800 billion per year. However, many non-indigenous species cause environmental damage and economic losses. In the United States, the annual cost of invasive species (including plants and aquatic organisms) is estimated at more than \$138 billion.^[7]

About 20 species of mammals have become established in the United States, including dogs (*Canis familiaris*), cats (*Felis catus*), cattle (*Bos taurus*), sheep (*Ovis aries*), horses, burros, pigs, goats [*Capra hircus*], and deer (*Cervus* spp.). Horses and burros introduced into western states number over 50,000 animals, which overgraze vegetation and decrease food for native animals. The Bureau of Land Management spends about \$22 million annually to manage these animals. Feral pigs cause damages nationwide of about \$800 million/year.^[7] Feral dogs cause about \$9–10 million in losses to cattle and sheep each year^[1,7] and feral cats kill about 465 million birds per year at an estimated cost of \$14 billion.^[7] Invasive mammals cause large agricultural losses. Nutria are pests in 15 states, causing over \$6 million per year^[1] in damage to sugarcane. Rat destruction of stored grains in the U.S. averages more than \$19 billion per year.^[7] Worldwide, rats are serious pests at farms, industrial sites, and homes. New Zealand spends over \$30 million annually controlling brushtail possums (*Trichosurus vulpecula*), which degrade native forests and spread tuberculosis,^[2] and \$2 million annually on feral goat control; cost figures do not include damage to forests and endangered species or reduced trade because of disease. About 97 of 1000 bird species in the U.S. are non-native; 5% of these, including chickens (*Gallus domesticus*), are considered beneficial while 56% are considered pests. Hawaii alone has 35 introduced species. The pigeon is the most serious pest bird in the U.S., with yearly damages estimated^[7] at \$1.1 billion to property and agricultural crops; pigeons can also spread over 50 human and livestock diseases.^[8] House sparrows, introduced into the U.S. to control canker worms, are now pests because they consume agricultural crops and ornamentals, displace native birds from nesting sites, and can spread 29 human and livestock diseases.^[8] European starlings are agricultural pests on grain and fruit crops, consume or contaminate livestock feed at feedlots, and are implicated in the spread of 25 diseases.^[8]

About 53 amphibian and reptile species in the U.S. are introduced, all in southern states and Hawaii.^[7] The brown tree snake was accidentally introduced on

the U.S. territory of Guam after World War II with military cargo. Snake populations 30 years later reached densities of 100/ha and caused the extinction of 10 of 13 native forest birds, 2 of 3 native mammals, and 9 of 12 native lizards. Snakes also cause frequent power outages by shorting out utility lines, resulting in \$1 million damage yearly.^[7] The cost to control snakes on Guam and limit their dispersion to other parts of the Pacific is estimated at \$6 million per year.

Invasive species can change ecosystems through their effects on vegetation. Introduced rabbits now dominate Australia and large parts of New Zealand, where they degrade habitats for native species and for livestock grazing. Feral pigs introduced into U.S. states for hunting now number about 4 million and damage both crops and the environment.

Invasive species have caused the extinction or endangerment of numerous native species throughout the world. Introduced rats and other mammalian predators are the major cause (42%) of bird extinctions on islands, with 54% attributed to rats, 26% to cats and the remainder to mongooses, weasels (*Mustela nivalis*), stoats (*Mustela erminea*), and other species like goats and pigs. About 42% of the almost 1000 species listed under the U.S. Endangered Species Act are at risk because of invasive species.^[9] In other world regions, about 80% of the endangered species are threatened due to non-native species. Rats have caused numerous extinctions; on Big South Island, a predator-free New Zealand refuge, a 1964 rat irruption eliminated five bird and one bat species. The mongoose is a classic case of biological control run amok. Beginning in 1872, it was introduced into Jamaica, Puerto Rico, other West Indian Islands, and Hawaii for control of rats in sugarcane; it preyed heavily on native reptiles, amphibians, and ground nesting birds, causing extinction and endangerment of many species.

Mating or competition between introduced and native species can lead to extinctions.^[4] Mallards (*Anas platyrhynchos*) introduced to Hawaii and Florida for hunting hybridized extensively with the endangered Hawaiian duck (*Anas wyvilliana*) and the Florida mottled duck (*Anas fulvigula*), threatening their existence. In the U.S., the introduced starling and house sparrow outcompete native songbirds, leading to a long-term decline in songbird species.

Introduced species also propagate diseases. In Hawaii, introduced Asian songbirds are host to avian pox and avian malaria,^[4] which have contributed to the elimination of many native birds. Small rodents introduced worldwide act as vectors of salmonellosis, leptospirosis, plague and murine typhus. Feral pigs spread brucellosis, pseudorabies, and trichinosis and the mongoose is a vector for rabies and leptospirosis in Puerto Rico and other islands.^[7]

MANAGEMENT OF INVASIVE SPECIES

The best method for dealing with invasive species is to prevent introductions. Although global traffic volume increases continuously, many nations have no invasive species policies. In the U.S., no comprehensive law addresses imports of non-native species, so regulatory agencies have often assumed a species will pose no problems unless proven otherwise.

It is more expensive to deal with introduced species once they are established than to prevent their introduction. Eradication is often the most cost-effective and ecologically sound solution, but can be difficult and sometimes controversial, and is most feasible in the early stages of invasion or on small islands. Rats have been eradicated using rodenticides on a number of areas, including the Aleutian Islands, Caribbean islands, and islands off New Zealand. Eradication of goats has been successful on 37 islands (up to 46,000 ha) throughout the world, primarily in New Zealand, Australia, and the Galapagos.

Where eradication is impossible, invasive species can often be managed to reduce their economic and/or ecological damage. Hunting can reduce the populations of feral pigs, feral goats, and Axis deer (*Axis axis*). Exclusion by fencing is successful but expensive. Trapping is used successfully to manage some invasive species. Brown treesnakes are trapped around airfields and ports on Guam to prevent their dispersal to other islands. Trapping and snaring are used to reduce pig and goat populations in Hawaii and in the Great Smoky Mountains National Park in the southeastern U.S.

Toxicants can provide a rapid initial reduction of invasive populations. Various rodenticides have been used effectively to manage rat populations throughout the world. Compound 1080 is used in Australia and New Zealand for controlling rabbit populations, in New Zealand for controlling brushtail possums, and on Aleutian and Pribolof Islands for eradicating arctic fox. A variety of toxicants have been used to control pigs, deer and goats. Acetaminophen (a human pain relief medicine) is being used to control or kill brown treesnakes on Guam, and caffeine sprays are being developed for controlling introduced frogs on Hawaii.

Research is being conducted on reproductive controls which could eventually reduce invasive species populations. Reproductive control will be most effective in managing species like rats with high reproductive and low survival rates, and least effective for species such as deer with low reproductive and high survival rates.

REFERENCES

1. Bergman, D.; Chandler, M.; Locklear, A. The economic impact of invasive species to wildlife services' cooperators. In *Human Wildlife Conflicts: Economic Considerations*, Fort Collins, CO, August 1–3, 2000; Clark, L., Hone, J., Shivik, J., VerCauteren, K., Watkins, R., Yoder, J., Eds.; Colorado State University Press: Fort Collins, CO, 2002.
2. Veitch, C.R. Management of exotic vertebrates: some of the New Zealand experience. *Proc. Vertebr. Pest Conf.* **2000**, *19*, 154–159.
3. Groves, R.H., Burdon, J.J., Eds.; *Ecology of Biological Invasions*; Cambridge University Press: Cambridge, U.K., 1986; 120–136.
4. Simberloff, D. *Impacts of Introduced Species in the United States*; <http://www.gcario.org/CONSEQUENCES/vol2no2/article2.html> (July 1999).
5. Brown, J.H. Patterns, modes and extents of invasions by invertebrates. In *Biological Invasions: A Global Perspective*; Drake, J.A., Mooney, H.A., di Castri, F., Groves, R.H., Kruger, F.J., Rejmanek, M., Williamson, M., Eds.; Wiley: Chichester, U.K., 1989; 85–109.
6. Fox, M.D.; Adamson, D. The ecology of invasions. In *A Natural Legacy: Ecology in Australia*; Recher, H.F., Lunney, D., Dunn, I., Eds.; Pergamon Press: New York, 1979; 135–151.
7. Pimentel, D.; Lach, L.; Zuniga, R.; Morrison, D. Environmental and economic costs of nonindigenous species in the United States. *BioScience* **2000**, *50*, 53–65.
8. Weber, W.J. *Health Hazards from Pigeons, Starlings and English Sparrows: Diseases and Parasites Associated with Pigeons, Starlings, and English Sparrows Which Affect Domestic Animals*; Thomson Publications: Fresno, CA, 1979; 1–138.
9. Nature Conservancy. *America's Least Wanted: Alien Species Invasions of U.S. Ecosystems*; The Nature Conservancy: Arlington, VA, 1996; 31 pp.

Mosquitoes: Biology

Daniel L. Kline

CMAVE, United States Department of Agriculture (USDA-ARS), Gainesville, Florida, U.S.A.

INTRODUCTION

Mosquitoes are the most prominent of the numerous species of blood-sucking arthropods that annoy man and other warm-blooded animals.^[1] They are true flies (Diptera) in the family Culicidae. Like all adult true flies, they have two wings.^[2] The most obvious characteristics separating adult mosquitoes from all other Diptera are a combination of wings with minute scales and female mouthparts, which form an elongate piercing-sucking proboscis.^[1] Males differ from females by usually having feathery antennae and mouthparts not suitable for piercing skin. While vertebrate blood is the primary food source for the females of most species, females often feed on flower nectar and various plant juices. Nectar is the principal food source of the males. Mosquitoes occur in practically every region of every continent in the world except Antarctica. They develop in an extremely broad range of biotic communities: arctic tundra, boreal forests, salt marshes, and ocean tidal zones. Many species have benefited from human alteration of the environment, and a few have become domesticated. Mosquitoes can be an annoying, serious problem in the human domain. They interfere with work and spoil hours of leisure time. Their attacks on farm animals can cause loss of weight and decreased milk production. Some mosquitoes are capable of transmitting disease organisms that cause malaria, lymphatic filariasis, yellow fever, and dengue to man, encephalitis to man and horses, and heartworm to dogs.^[3]

CLASSIFICATION AND RECOGNITION

Culicidae consists of about 3200 recognized species. Current culicid classification recognizes three subfamilies: Anophelinae, Culicinae, and Toxorhynchitinae. There are 38 genera of mosquitoes, 34 of which are in the subfamily Culicinae. Culicines are organized into 10 tribes, the most diverse of which are Aedini and Sabethini in terms of numbers of genera and species worldwide.^[3]

Three important species groups of mosquitoes worldwide are the *Anopheles gambiae* and *Culex pipiens* complexes and the *Aedes* subgenus *Stegomyia*. The *A. gambiae* complex in Africa consists of six species.

Two of these, *A. gambiae* and *Anopheles arabiensis*, are important vectors of malaria and lymphatic filariasis. Both prefer to bite humans, but *A. gambiae* lives in close association with humans, and therefore is the more important vector. The *C. pipiens* complex is a ubiquitous group of closely related domestic and peridomestic species. The medically most important taxa worldwide are the temperate species *C. pipiens*, the northern house mosquito, and the tropical and subtropical *Culex quinquefasciatus*, the southern house mosquito. Their ranges are overlapping in the central latitudes of the U.S.A., where they commonly hybridize. They are vectors of several human pathogens, such as St. Louis encephalitis virus, West Nile virus (WNV), and worms that cause filariasis. Several brightly marked *Aedes* species in the large subgenus *Stegomyia* are medically important, including *Aedes aegypti* and *Aedes albopictus*. *A. aegypti*, the yellow fever mosquito, has a worldwide distribution in the tropics and subtropics. It is the primary vector of both dengue and urban yellow fever viruses. *A. albopictus*, the Asian tiger mosquito, is similar to *A. aegypti*, occupies the same kinds of containers, and also transmits dengue virus. A cold-hardy, egg-diapausing strain of this mosquito has been carried from northern Japan to other parts of the world by the trade in used automobiles and truck tires. While in most of its range in the southern U.S.A. *A. albopictus* has replaced *A. aegypti* as the predominant pest mosquito species, worldwide *A. aegypti* is more important owing to its role in virus transmission.^[3]

LIFE CYCLE

Mosquitoes undergo what is called complete metamorphosis, a very complicated life cycle that involves four distinct stages: egg, larva, pupa, and adult. Each of these stages can be easily recognized by their special appearance. The larval and pupal stages only occur in water, and adults emerge from pupae and move to land.^[2,4]

Although some mosquitoes lay their eggs on the surface of the water, others deposit their eggs on moist surfaces such as soil or in containers (artificial and natural). Eggs are either deposited singly or as an egg raft depending on the type of mosquito. Eggs laid on

moist soil may lie dormant for several months or even years before hatching. Hatching occurs when the eggs are covered by rainwater or tides. Larvae will emerge from eggs within two to three days when environmental conditions are ideal. All larvae go through four developmental stages called instars. This process usually requires four to five days depending upon environmental conditions such as temperature and food availability. The range is three days for some species of *Psorophora* in the tropics to greater than one year for some Arctic *Culiseta*. Most larvae have siphon tubes for breathing and hang from the water surface. *Anopheles* larvae do not have a siphon and lie parallel to the water surface to get a supply of oxygen through a breathing opening. *Mansonia*, *Coquillettidia*, and some *Mimomyia* species are unusual in remaining submerged throughout larval and pupal development, with their siphons embedded in the tissues of aquatic plants from which they derive some oxygen. The larvae feed on microorganisms and organic matter in the water. On the fourth molt the larva changes into a pupa. The pupal stage is a resting, non-feeding stage. This is the time the mosquito turns into an adult. It takes about two days before the adult is fully developed; the pupal skin splits at the water's surface and the adults emerge. Males usually emerge a day before the females. Shortly after the females emerge they mate. After three to four days, only the females are ready to bite. The females seek a blood meal to obtain the protein necessary for the development of her eggs. With one blood meal, a female may produce 250 or more eggs. After a blood meal, it takes three to five days for the blood to be digested and the eggs to develop. Females may produce two to four egg batches. Females are capable of transmitting pathogens if they live long enough for the pathogens to multiply and/or develop within their bodies between blood meals.^[2-4]

HOST PREFERENCE

Adult mosquitoes of both sexes regularly feed on sugar sources such as plant nectar and honey dew throughout their life, but only females feed on vertebrate blood. Many species are specific in their host preference for birds, mammals, or cold-blooded vertebrates such as reptiles and frogs. Consequently, various mosquito species use a wide variety of cues to find a suitable host, often involving a variety of complex interactions, which are still not fully understood. It is known that host-finding behavior in mosquitoes involves the use of volatile chemicals to locate vertebrate hosts. Several hundred compounds found in human breath, secretions, and sweat glands have been identified and vary in their degree of attractiveness to

female mosquitoes. Carbon dioxide, lactic acid, and octenol are among the best-documented host attractants.^[2,5,6] Other skin emanations also are known to be important, because odors from live hosts are always more attractive than any combination of these chemicals in a warm, humid airstream.^[3,5] Fatty acids produced by the normal bacterial flora of the skin are particularly effective in attracting *A. gambiae* to human feet. Mixtures of these fatty acids probably play a major role in attracting most mosquitoes. Subtle differences in these odors of different host species and even different individuals undoubtedly play a role in host preference. These odors commonly have a combined effective range of 7–30 m, but the range can be up to 60 m for some species.^[3] Vision also is important in orienting to hosts, particularly for diurnal species, and especially in an open environment and at intermediate or close ranges. Dark, contrasting, and moving objects are particularly attractive. As a female approaches to within 1–2 m of a potential host, chemical and visual cues are still important, but convective heat and humidity surrounding the body also come into play. Odor, carbon dioxide, heat, and humidity all are detected by sensilla on the antennae and palps.^[3,5,6]

PUBLIC AND VETERINARY HEALTH IMPORTANCE

Mosquitoes are of public health significance because they feed on human blood. Blood feeding compromises the skin, presenting the possibility of secondary infection with bacteria. Females introduce foreign proteins with saliva that stimulate histamine reactions, causing localized irritation that may be antigenic, leading to hypersensitivity, and allowing for acquisition and transmission of microorganisms that cause infection and disease in humans, domestic animals, and wild animals. Mosquito-borne diseases are caused by three groups of pathogens: viruses, protozoans (malarial), and filarial nematodes. In addition to the tremendous impact of mosquitoes on human health as vectors of disease pathogens, the bites themselves are important. Aside from the annoying flight and buzzing sound, a single bite can be irritating and a distracting nuisance.

As with other blood-feeding arthropods, the wound created at the bite site may allow secondary infection by bacteria, which can be exacerbated by scratching.^[3]

Mosquitoes are also important as vectors of disease agents to animals. Mosquito-borne viruses affecting domesticated animals include the groups of alphaviruses that are associated with the eastern, western, and Venezuelan equine encephalitides, and the flavivirus WNV, all of which cause an acute encephalitis with high fever in equids (horses, donkeys, and mules).

Other mosquito-borne viruses of veterinary significance include Japanese encephalitis virus, Rift Valley fever virus, Wesselsbron virus, fowlpox virus, and myxomatosis virus. Many *Plasmodium* species infect animals other than humans, including reptiles, birds, rodents, and nonhuman primates. Dog heartworm is caused by the mosquito-borne filarial nematode *Dirofilaria immitis*. Aside from their importance as vectors of disease agents in animals, mosquitoes are a cause of irritation, blood loss, and allergic reactions. They not only annoy, but also disrupt normal behavior of livestock and companion animals. Large swarms may cause livestock to discontinue feeding and seek relief. Increased scratching behavior may result in skin abrasions, hair loss, and secondary infection with bacteria at the bite and scratch sites. For cattle, mosquito bites can result in decreased weight gains and milk production, and prompt producers to alter pasturing practices. Deaths of cattle owing to anemia and stress have been reported.^[3]

CONCLUSIONS

Mosquitoes occur worldwide. There are over 3200 species. The life cycle consists of egg, larva, pupa, and adult. Development takes place in a broad range of

biotic communities. Only the adult female bites. In addition to annoyance, mosquito bites can lead to secondary infection, allergic reactions, or transmission of pathogens to humans and livestock, such as viruses, protozoans, and filarial worms.

REFERENCES

1. James, M.T.; Harwood, R.F. Mosquitoes. In *Herm's Medical Entomology*, 6th Ed.; The Macmillan Company: Toronto, 1969; 167–222.
2. Anderson, R.R.; Harrington, L.C. *Mosquito Biology for the Homeowner*; <http://www.entomology.cornell.edu/MedEnt/MosquitoFS/MosquitoFS-print.html> (accessed April 15, 2005).
3. Foster, W.A.; Walker, E.D. Mosquitoes (Culicidae). In *Medical and Veterinary Entomology*, 1st Ed.; Mullen, G.A., Durden, L., Eds.; Academic Press: New York, 2000; 203–262.
4. Anonymous. *Mosquito Biology*; http://www.pasco-mosquito.org/mosquito_biology.htm (accessed May 11, 2005).
5. Clements, A.N. *The Biology of Mosquitoes. Vol. 2. Sensory Reception and Behaviour*; CABI Publishing: New York, 1992.
6. Kline, D.L. Attractants for mosquito surveillance and control: a symposium. *J. Am. Mosq. Control. Assoc.* **1994**, *10* (2), 253–338.

Mosquitoes: Control

Daniel L. Kline

CMAVE, United States Department of Agriculture (USDA-ARS), Gainesville, Florida, U.S.A.

INTRODUCTION

Mosquito control can be divided into two areas of responsibility: individual and community. Individual responsibility includes personal protection measures (e.g., repellents), space sprays, and source reduction. Many mosquito problems cannot be controlled by individuals, but need to be managed through an organized community effort because mosquitoes do not recognize property boundaries.

INDIVIDUAL EFFORTS

Personal protection is the most direct and simple approach to prevent mosquito bites by individuals. Exposure to mosquito bites can be minimized by staying indoors during peak mosquito activity periods, wearing protective clothing such as long sleeve shirts, long pants, socks, and shoes, and/or using repellents. Chemical repellents applied to skin or clothing prevent mosquitoes from landing or cause them to leave before probing. Two common synthetic repellents are DEET (*N,N*, diethyl-3-methyl benzamide) and permethrin. DEET can be applied directly to the skin or clothing. Permethrin, an insecticide with repellent properties, should only be applied to clothing. Commercially available head nets and permethrin-treated clothing are now available for use by homeowners. Head nets reduce annoyance and prevent bites about the face and neck. In developing countries, bed nets, impregnated with synthetic pyrethroids and strung over beds at night, repel mosquitoes and kill those that land on the nets.^[1,2]

Repellents are formulated and sold as aerosols, creams, solids (sticks), and liquids. Multiple concentrations and formulations of DEET are readily available. Multiple chemical, botanical, and “alternative” repellent products are also marketed to consumers. In a recent study, the efficacy of seven botanical insect repellents, four products containing DEET, and a repellent containing IR3535 (ethyl butylacetylaminopropionate) were tested. DEET-based products provided complete protection for the longest duration. Higher concentrations of DEET provided longer lasting protection. A formulation containing 23.8% DEET had a mean

complete-protection time of 301.5 min. A soybean oil-based repellent protected against mosquito bites for an average of 94.6 min. The IR3535-based repellent protected for an average of 22.9 minutes. All other botanical repellents tested provided protection for a mean duration of less than 20 minutes. Repellent-impregnated wristbands offered no protection.^[2,3] Recently published research on the comparative evaluation of IR3535 and DEET on an equal formulation (cream and liquid)/concentration (10% and 20%) basis showed that both active ingredients performed similarly against two species of mosquitoes.^[4]

Other devices create a repellent smoke or vapor that reduces mosquito attack in the immediate vicinity. Dispensers of citronella or essential oils (e.g., linalool or geraniol) that conceal human odors can be used where humans congregate to give added protection. Citronella candles and torches are most useful outdoors under calm air conditions. Their effectiveness is considerably less than repellents applied to the body or clothing.

Space sprays may be used to kill mosquitoes present at the time of treatment. Homeowners may use handheld foggers or fogging attachments on tractors or lawn mowers for temporary relief from flying mosquitoes. Pyrethrins or 5% malathion can be fogged outdoors. Mosquitoes can be killed inside the house by using a household aerosol space spray containing synergized pyrethrum or synthetic pyrethroids (allethrin, resmethrin, etc.). Only insecticides labeled for flying insect management should be sprayed into the air. Best results are obtained if doors and windows are kept closed during spraying and for 5–10 min after spraying. The major advantage of space treatment is immediate knockdown, quick application, and relatively small amounts of materials required for treatment. Space sprays are most effective indoors. Outdoors, the insecticide particles disperse rapidly and may not kill many mosquitoes. The major disadvantage of space spraying is that it will not manage insects for long periods of time.^[5]

Homeowners can also reduce mosquito numbers in their backyards by practicing source reduction. They can destroy or dispose of tin cans, old tires, buckets, plastic sheeting, or other containers that collect and hold water; keep water from accumulating at the base

of flower pots or in pet dishes for more than two days; change water in bird baths and wading pools at least once a week and stock ornamental pools with top feeding predaceous minnows (known as mosquito fish, these minnows are about 1–1.5 in. in length and can be purchased or seined from streams and creeks); fill or drain puddles, ditches, and swampy areas, and remove, drain, or fill tree holes and stumps with mortar; eliminate seepage from cisterns, cesspools, and septic tanks; eliminate standing water around animal watering troughs; and irrigate lawns and gardens carefully to prevent water from standing for several days.^[5,6]

Homeowners can also practice vegetation management. Adult mosquitoes prefer to rest on weeds and other vegetation. Homeowners can reduce the number of areas where adult mosquitoes can find shelter by cutting down weeds adjacent to the foundation and in their yards, and mowing the lawn regularly. To further reduce adult mosquitoes harboring in vegetation, insecticides may be applied to the lower limbs of shade trees, shrubs, and other vegetation. Products containing allethrin, malathion, carbaryl, or chlorpyrifos have proven effective. Paying particular attention to shaded areas, insecticides should be applied as coarse sprays onto vegetation, walls, and other potential mosquito resting areas using a compressed air sprayer.^[5]

Many people are reluctant to use repellents or pesticides. They prefer to use traps that have recently become commercially available. Most commercial traps use attractants that are targeted at mosquitoes and other biting flies; beneficial insects are spared. Carbon dioxide (CO₂) is used as the primary attractant. CO₂ can be generated by catalytic burning of propane, released from a compressed gas cylinder, generated chemically or photocatalytically. Some traps utilize excess heat from the combustion of propane to fuel a thermoelectric generator to power fans that suck mosquitoes into a collecting container; no batteries or main line current are needed. Heat and moisture are produced simultaneously, and these are also good mosquito attractants. Electricity produced by propane combustion allows these traps to be portable for use in remote areas. About 20 lbs of propane generates 60 lbs of CO₂ and lasts about three weeks in continuous operation. Mosquitoes attracted to the traps are usually captured by fans that pull them into a net, sticky trap, catch basin, or electric grid. Other traps utilize main line current and CO₂ from gas cylinders. Manufacturers of these traps claim that releases of CO₂ from cylinders are easier to program. Some traps also use octenol, UV light, and programmed flashing of multicolored light emitting diodes as attractants. One trap emits the sound of a dog heartbeat as its primary attractant. Many models of commercial traps are now available

with different configurations being produced each year.^[7,8]

Trap placement is one of the keys to success with these traps. To be effective, traps should be placed between mosquito breeding areas and areas where people will congregate. Mosquitoes should encounter the trap before they detect people. Traps should be placed upwind from human activities, preferably in shady, open locations.

Studies indicate that these traps definitely capture large numbers of mosquitoes. What remains to be determined is whether these traps can successfully reduce backyard mosquito populations? Or better yet can they reduce the number of bites? So far, there is a lot of anecdotal evidence that they do indeed reduce mosquito nuisance in backyards to a tolerable level, but scientific confirmation is lacking.

Traps should not be considered magic bullets that destroy all the biting mosquitoes by themselves. They should be considered as one part of an integrated pest management (IPM) program that includes source reduction (elimination of larval breeding sites), vegetation management, space sprays, and the judicious use of repellents. Repellents would probably be effective longer, if the mosquito density was lowered by traps. Traps would be more effective if hosts were disguised by repellents.^[8]

ORGANIZED COMMUNITY CONTROL

Management of mosquito problems often requires area-wide control by county-level mosquito abatement districts, which often utilize an IPM approach. Larvae are often targeted because they tend to be concentrated in relatively small areas. Permanent larvae control measures used include impounding water, ditching, or draining swampy breeding areas. Temporary measures include treating developmental sites with chemical insecticides. Currently, categories of registered larvicides are light mineral oils, organophosphates, and insect-growth regulators. The insect-growth regulator, methoprene, is a mimic of juvenile hormone and interferes with metamorphosis and emergence. Biological control of larvae by predators or pathogens has been studied extensively, but operational success has been limited. An exception is the bacterium *Bacillus thuringiensis israelensis*, or *Bti*, which has been developed into commercial formulations since its original discovery in 1975. It is used extensively in mosquito control programs. Larvae die when they ingest crystalline, proteinaceous toxins produced by the bacterial cells during sporulation. The bacterium, *Bacillus sphaericus* has a similar mode of action but is more specific. It is particularly effective against *Culex* larvae,

and it is more persistent in water and more tolerant of water with a high organic content than is *Bti*.^[5,6]

Because adult mosquitoes can fly long distances, it is often necessary to supplement larval control with control measures directed against adult mosquitoes— aerosol spraying of insecticides by ground or aerial equipment to kill adult mosquitoes. Adulticides intended for direct contact between airborne droplets and mosquitoes are of two types: thermal fogs and low- or ultralow-volume sprays. Both can be applied from hand-carried equipment, motor vehicles, or aircraft. Currently, insecticides registered for use in fogs and low-volume sprays are organophosphates (e.g., temephos, malathion, and chlorpyrifos), carbamates, pyrethrins, and synthetic pyrethroids. Adulticides are also applied to surfaces where adults will rest or in the air where they fly. Residual insecticides applied to resting surfaces may retain their toxicity for days to months. Residual adulticides also can be used outdoors on vegetation or structures that serve as harborage. They tend to have short-term effects, because sunlight, wind, and rain cause the insecticide to degrade. Resistance to insecticides is an important consequence of their use and has developed in many mosquito populations.^[5,6]

In developing countries, there is now increasing emphasis on community cooperation, low technology, sustainability, and the integrated use of a variety of control tools that are adapted to local customs, conditions, and resources.

Genetic control, a biological control category using a variety of genetic methods, has been successful against some pests; however, its use against mosquito vectors of disease remains experimental. There is a lot of research being conducted on utilizing transgenic mosquitoes to develop strains of mosquitoes that do not bite humans or are incapable of transmitting disease agents, but practical use is in the distant future.^[5]

CONCLUSIONS

Four overlapping objectives of mosquito control are to prevent bites, reduce mosquito populations to acceptable densities, minimize mosquito–vertebrate contact, and reduce the longevity of female mosquitoes. If these objectives are successfully accomplished, then annoyance and the risk of obtaining mosquito-borne diseases will be reduced. Individuals can obtain some relief using personal protection methods and practicing source reduction around their homes, but effective, sustainable, community-wide reduction in mosquito-associated problems requires organized efforts.^[5]

REFERENCES

1. Catangui, M.A.; Wilson, J.A. Personal mosquito repellents, 2003; <http://agbiopubs.sdstate.edu/articles/FS920.pdf> (accessed March 2005).
2. Fradin, M.S.; Day, J.F. Comparative efficacy of insect repellents against mosquito bites. *N. Engl. J. Med.* **2002**, *347*, 13–8.
3. Anon. The buzz on repellents. *Consumer Reports*; May 2003; 15 pp.
4. Cilek, J.E.; Petersen, J.L.; Hallmon, C.F. Comparative efficacy of IR3535 and DEET as repellents against adult *Aedes aegypti* and *Culex quinquefasciatus*. *J. Amer. Mosq. Control Assoc.* **2004**, *20*, 299–304.
5. Foster, W.A.; Walker, E.D. Mosquitoes (Culicidae). In *Medical and Veterinary Entomology*; Mullen, G.A., Durden, L., Eds.; Academic Press: New York, 2000; 203–262.
6. University of Florida, Florida Coordinating Council on Mosquito Control. Florida mosquito control: the state of the mission as defined by mosquito controllers, regulators, and environmental managers, 1998.
7. Anon. Should you trap or zap? *Consumer Reports*; May 2003; 16–17.
8. Quarles, W. Mosquito attractants and traps. *Common Sense Pest Control* **2003**, *29* (2), 4–14.

Mosquitoes: Human Attacks

Eric J. Hoffman
Edward D. Walker
James R. Miller

Michigan State University, East Lansing, Michigan, U.S.A.

INTRODUCTION

Mosquitoes are important vectors of disease agents and are a worldwide nuisance to vertebrate animals as well. Mosquito-borne diseases (viruses, protozoa, and nematodes) result in millions of human infections and deaths yearly, primarily in tropical areas, but also in subtropical and temperate regions. In 2003, there were some 9000 reported human cases of West Nile viral meningoencephalitis and West Nile fever in the United States and 244 deaths. In the tropics, malaria remains the most important vector-borne disease of humans. According to the UN Roll Back Malaria program, malaria contributes to up to a 1.3% reduction in economic growth in some African countries. The disabilities and deformities resulting from mosquito-borne filariasis (such as elephantiasis) are well known; hundreds of millions of people are chronically infected in tropical areas. The emergence of pathogenic forms of mosquito-borne diseases, such as dengue hemorrhagic fever and dengue shock syndrome, emphasizes the dynamic nature of mosquito-borne disease systems and the difficulty in controlling them without concerted, well-funded programs. Infections in domestic animals and wildlife due to mosquito transmission are important causes of morbidity and mortality in these species.

Approaches to mosquito control range from reduction in number or quality of larval habitats, to treatment of larval habitats with larvicidal materials, to antiadult measures, and to measures to prevent bites. These activities can range from protection of an individual person to areawide management for whole communities. Personal protection includes such measures as repellent and insecticide-treated clothing, bednetting, and application of chemical repellents such as DEET. More sophisticated management programs include regional survey of larval and adult mosquito populations followed by appropriate intervention measures such as drainage of breeding sites, treating water to kill larvae and pupae, or using insecticides to kill adults.

BIOLOGY AND BEHAVIOR

Although adult, flying mosquitoes are most familiar, the mosquito life cycle is intrinsically tied to aquatic environments. In general, eggs are deposited in or near water where the larvae hatch and feed on suspended nutrients, bacteria, and other organic matter. Habitats for the immature stages vary greatly by species (e.g., puddles, tree holes, ponds, lakes, and rivers). Pupation is also completed in the water. Most adult females require vertebrate blood for provision of their eggs. Mammals, birds, reptiles, and amphibians all serve as hosts for mosquito bloodfeeding, and many mosquito species prefer particular hosts within these groups. Some adult females can fly several kilometers in an evening of foraging for blood.^[1,2]

Mosquito orientation to humans is mediated by short-range cues such as body heat and moisture, as well as long-range ones such as odor and visual cues. Carbon dioxide (CO₂) is a potent attractant of most mosquito species. L-lactic acid can also be an important mediator for hostseeking.^[3] Metabolites from microbes associated with the human body can enhance the attractiveness of humans to mosquitoes. Bacterially produced foot odor is an attractant of the malaria mosquito, *Anopheles gambiae*^[4] and explains the preference of this species for biting around the feet.

Mosquito adults are intolerant of air that is hot and dry.^[5] Accordingly, adults usually forage for hosts and ovipositional sites at dawn, dusk, and night when wind velocities are low and humidity is higher. Until recently, it was thought that mosquito adults do not forage in appreciable winds because they were too weak to make headway into the wind. However, Hoffmann and Miller^[6] found that the effect of wind in reducing mosquito hostfinding is best explained by dilution of the attractants emanating from hosts.

REGIONAL MANAGEMENT

Efforts to control mosquitoes at the single, backyard scale often have limited impact through time.

Mosquitoes from nearby unmanaged areas can reinvade in just a few days after a backyard is fogged with an insecticide. The most effective mosquito management programs are coordinated at a regional scale (500–5000 square miles) that is considerably larger than the flight range of these pests.

Monitoring

Monitoring is a cornerstone of regional mosquito management. It addresses several interrelated questions: Which species are present? How numerous or dense are they? From where are they coming? Combined with assays for the presence of disease organisms, mosquito surveys answer the critical question: How dangerous is a bite?

Monitoring of adult mosquitoes is accomplished by deploying baited traps at sites of interest, including those with a history of significant infestations. The most common trap type for hostseeking mosquitoes uses an attractant light and/or CO₂ with a small electric fan that blows individuals into a collecting bag (Fig. 1). Landing rates and bite counts on human subjects are also used to assess population levels (Fig. 2). Gravid traps are aimed at females ready to oviposit. These devices are containers of aged water



Fig. 1 A Centers for Disease Control (CDC) light trap with a container to release dry ice (CO₂). (Photo by Eric J. Hoffmann.)



Fig. 2 Researchers taking bite counts in a Michigan wetland. (Photo by Eric J. Hoffmann.)

and organic matter (e.g., hay infusion) emitting volatiles attractive to gravid females. Larvae from eggs laid in ovitraps are identified to species and counted as a measure of localized density.

Effective mosquito monitoring programs also sample larval/pupal habitats. This typically involves dipping a defined volume of water from a habitat, and identifying and counting the mosquito contents. The most successful mosquito management programs focus, primarily, on larval populations and, secondarily, on adult populations.

Monitoring for mosquito-borne disease is accomplished by using sentinel animals, such as chickens. Serum samples can be tested for viral antibodies or nucleic acids. Sick or dead animals such as horses and birds can be unintended sentinel animals if a program is in place to test them. The movement of West Nile virus across the United States has been tracked largely by analysis of dead birds. Mosquitoes themselves can be analyzed for pathogens; mosquito catches in traps can be pooled by region and analyzed with polymerase chain reaction (PCR) techniques to detect specific viral RNA.

Controls

Reducing larval habitats by draining or filling bodies of standing water is a cornerstone of mosquito control. This practice is highly recommended and uncontroversial when applied to containers discarded by humans (e.g., buckets, toys, and tires). But many bodies of water have important functions both for human and natural systems. For example, catch basins and storm sewers cannot be eliminated, and wetlands support a rich diversity of important plant and animal life, whose welfare must be balanced against that of humans.

Insecticides are available for treating such pools of water and targeting mosquito larvae. These chemistries

are not perfect solutions, but can reduce mosquito pressure in many situations. Formulations of the bacteria *Bacillus thuringiensis israelensis* (*Bti*) and *Bacillus sphaericus* are widely utilized for larval control. Larvae ingest the bacteria and associated toxins then bind specifically to receptors of midgut cells. Death results from disruption of the gut. Insect growth regulators such as Methoprene,^[7] or surface oils, are also useful against immature stages. Temephos, an organophosphate, has been used for larval mosquito control since 1965. This chemical can be very effective against mosquitoes and, according to the World Health Organization, is not harmful to humans or animals when used at labeled doses, including in drinking water.

In certain cases, the nuisance level or risk of contracting a disease carried by mosquitoes is such that adult control measures are required. Insecticide barrier treatments include applications of residual formulations to vegetation and surfaces where mosquitoes rest. In some tropical settings, indoor residual sprays onto walls of domiciles are used. Insecticides incorporated into bednet materials greatly enhance the barrier offered by nets. Another approach is the application of insecticides in ultralow-volume formulation, in which concentrates of insecticides are applied at low rates into the air when mosquitoes are flying, either by hand equipment or from vehicles or aircraft. Formulations include chemicals in the organophosphate, carbamate, pyrethrin, and synthetic pyrethroid classes. All insecticides must be used in strict accordance with their labels.

Increasing populations of mosquito-eating birds, bats, and fish are sometimes promoted as an environmentally friendly and efficacious tactic in managing mosquitoes. Although creating habitat for these animals is admirable, the number of mosquitoes consumed by these predators has been drastically overestimated in popular literature.

Resistance Monitoring

Like other insects, mosquitoes have developed resistance to pesticides. Both biochemical and behavioral assays are used for screening populations for resistance.^[8] Along with monitoring for resistance, rotation among compounds with different modes of action is an important element for mosquito management.^[9]

PERSONAL PROTECTION

Even with regional management, mosquitoes often remain a sufficient nuisance such that individuals wish to take further steps for protection. There are many personal protection measures available to reduce the

number of bites and the associated risk of disease. Protective clothing is a first-line defense against mosquito bites (e.g., long sleeves, long pants, socks, and shoes).

Only a few highly effective chemical repellents are available for reducing bites by mosquitoes. Chief among them is DEET; *N,N*-diethyl-*m*-toluamide has been available since 1956 and is the most commonly used and broadly effective insect repellent in the world. Despite its proven efficacy, there is appreciable public concern about DEET's toxicity. This compound was extensively tested in a 1980 USEPA re-registration and was found safe when used according to label directions. Similar conclusions have come from current reviews of historical clinical data.^[10,11] The piperidines represent a promising class of repellents. Bayrepel® (Bayer AG) is currently being registered through the USEPA and FDA, and may rival DEET in effectiveness.^[12]

Insecticide-treated bednets (ITNs) have been extensively tested in Africa to reduce the burden of malaria and other mosquito-vectored diseases.^[13] If adopted regionally, ITNs can reduce the overall mosquito population and benefit non-users as well as net users.^[12]

Various devices are marketed to trap or otherwise kill mosquitoes in the backyard setting. Most emit combinations of light, CO₂, and heat to attract mosquitoes. Although these devices can catch/kill mosquitoes, their ability to significantly reduce the number of bites within the zone of use remains controversial. A challenge for such devices is that their sphere of influence covers only a small portion of a backyard at a given time. Mosquitoes may quickly and continually repopulate a backyard from surrounding sources.

CONCLUSION

Mosquito management is vitally important to human and animal welfare. Mosquito repellents can reduce biting, and protective clothing and barriers are important elements of managing mosquito exposure. However, the threat of disease is best reduced by managing mosquito breeding sites and activity on a regional scale.

REFERENCES

1. Clements, A. *The Biology of Mosquitoes*; Chapman and Hall: London, 1992; Vol. 1.
2. Clements, A. *The Biology of Mosquitoes*; Chapman and Hall: New York, 1992; Vol. 2.
3. Smith, C.N.; Smith, N.; Gouck, H.K.; Weidhaas, D.E.; Gilbert, I.H.; Mayer, M.S.; Smittle, B.J.; Hofbauer, A. L-Lactic acid as a factor in the attraction of *Aedes*

- aegypti* (Diptera: Culicidae) to human hosts. *Ann. Entomol. Soc. Am.* **1970**, *63*, 760–770.
4. Knols, B.G.J.; Takken, W.; Cork, A.; De Jong, R. Odour-mediated, host-seeking behaviour of *Anopheles* mosquitoes: a new approach. *Ann. Trop. Med. Parasitol.* **1997**, *91* (Suppl. 1), S117–S118.
 5. Platt, R.B.; Collins, C.L.; Witherspoon, J.P. Reactions of *Anopheles quadrimaculatus* say to moisture, temperature and light. *Ecol. Monogr.* **1957**, *27* (3), 303–324.
 6. Hoffmann, E.J.; Miller, J.R. Reassessment of the role and utility of wind in suppression of mosquito (Diptera: Culicidae) host finding: stimulus dilution supported over flight limitation. *J. Med. Entomol.* **2003**, *40* (5), 607–614.
 7. Ali, A.; Nayar, J.K.; Xue, R.-D. Comparative toxicity of selected larvicides and insect growth regulators to a Florida laboratory population of *Aedes albopictus*. *J. Am. Mosq. Control Assoc.* **1995**, *11* (1), 72–76.
 8. Brooke, B.D.; Hunt, R.H.; Koekemoer, L.L.; Dossou, Y.J.; Coetzee, M. Evaluation of a polymerase chain reaction assay for detection of pyrethroid insecticide resistance in the malaria vector species of the *Anopheles gambiae* complex. *J. Am. Mosq. Control Assoc.* **1999**, *15* (4), 565–568.
 9. Hemingway, J.; Penilla, P.R.; Rodriguez, A.D.; James, B.M.; Wedge, W.; Rogers, H.; Rodriguez, M.H. Resistance management strategies in malaria vector mosquito control: a large scale field trial in southern Mexico. *Pestic. Sci.* **1997**, *51* (3), 375–382.
 10. Osimitz, T.G.; Grothaus, R.H. The present safety assessment of DEET. *J. Am. Mosq. Control Assoc.* **1995**, *11*, 274–278.
 11. Goodyer, L.; Behrens, R.H. Short report: the safety and toxicity of insect repellents. *J. Trop. Med. Hyg.* **1998**, *59* (2), 323–324.
 12. Yap, H.H.; Jahangir, K.; Chong, A.S.C.; Adanan, C.R.; Chong, N.L.; Malik, Y.A.; Rohaizat, B. Field efficacy of a new repellent, KBR 3023, against *Aedes albopictus* (SKUSE) and *Culex quinquefasciatus* (SAY) in a tropical environment. *J. Vector Ecol.* **1998**, *23* (1), 62–68.
 13. The western Kenya insecticide-treated bed net trial. *Am. J. Trop. Med. Hyg.* **2003**, *68* (Suppl. 4).

Mulches and Pests

Lars Olav Brandsæter

Department of Herbolgy, The Norwegian Crop Research Institute,
Plant Protection Center, Ås, Norway

INTRODUCTION

With increasing knowledge about pesticides in the environment, growing public pressure to reduce pesticide use, and increased interest for integrated and organic pest management, growers require alternatives to chemical pesticide control. The use of artificial or organic mulches for soil surface covering will directly influence pests and is an interesting approach for pest management, particularly in row crops. Mulches include both artificial materials, such as plastic films or paper, and organic materials, such as straw, grass or clover clippings, sawdust, and animal manure. Both artificial and organic mulches have been tested in numerous studies, and many of these materials are in commercial use. In addition to direct effects on pests, mulches will also influence the performance of the crop, and the total effect of introducing mulch cropping systems will be the sum of effects, including the interactions, on the crop and the pests. Other cropping systems (e.g., some winter annual legume cover crop systems) are closely related to mulch systems. However, mulch systems are here defined only to include mulch materials produced from another place and transported into the field for ground covering.

In a hot and dry climate (tropical and subtropical), mulching offers additional benefits such as enhanced soil moisture and organic matter. In a temperate climate, however, the effect of decreased soil temperature, typical for some types of mulches, may be detrimental.

General mulch impacts weed, arthropods, and diseases, and examples of the use of mulches for pest management in vegetables and orchards are discussed below. Economical and technical considerations connected to this kind of pest management are briefly discussed.

INFLUENCE ON PESTS

Mulches can alter soil moisture, soil temperature and light conditions, soil texture, and nutrient availability. All these factors will affect crop performance and consequently influence the degree of competitive ability against weeds, tolerance to arthropods, and susceptibility to diseases. More specific mulch impacts on weeds, arthropods, and diseases are listed below.

Different types of mulches may prevent weed seed germination and seedling establishment by: 1) modifying the microclimate, including effects on light interception, the magnitude and the fluctuation in temperature, and moisture in soil; 2) creating a physical barrier to seedling growth; and 3) providing natural chemicals, often called allelochemicals, or substrates for production of allelochemicals by microorganisms. In natural environments, microclimate, physical barriers, and chemical effects interact, and it is difficult to separate single factors with regard to their effects on seed germination and seedling growth.

Regarding arthropod pests, mulching implies a manipulation of the agroecosystem affecting both the crop and the pest organisms, as well as the enemies of the pests. The complexity of mulch systems makes it difficult to draw clear conclusions, however, some general aspects can be listed: 1) Specific types of mulch alter the microclimate, making the habitat more or less favorable for herbivores. 2) Mulches may disturb herbivores' host-plant selection by changes in host plant density, naturally occurring attractant/repellent chemicals, or background (color) effects. 3) The use of organic mulch can influence the densities of different arthropod predators and parasitoids. 4) Epizootics of infectious insect diseases are influenced by environmental factors, and several studies have shown that habitat manipulation might enhance conditions for epizootic development. Depending on the arthropod/pathogen system, the use of mulch might enhance or inhibit an epizootic development.

Mulch effects are most commonly highlighted from the point of view of weeds and arthropods; however, mulch practice can also have a strong influence on the occurrence and epidemiology of diseases by: 1) altering the microclimate making the physical condition more or less favorable for different disease organisms; 2) providing alternative substrates; 3) influencing splash dispersal; 4) altering the behavior of vectors (e.g., aphids and thrips).

EXAMPLES

The effects of different mulches on pests have been tested in several row crops such as vegetables and fruits.

Vegetables

In vegetables, especially in tomato production, several mulch experiments have shown that black plastic film blocks weeds efficiently. Paper is another artificial mulch that can be used in vegetable production, but a problem with paper is quick biodegradation and hence a shorter weed effect than plastic film. Paper can be used in short-season crops like lettuce, and paper mulch does not need to be removed from the field after harvest. Moreover, different paper qualities are available and paper can be coated, e.g., with resins based on vegetable oils, to slow the rate of degradation. Experiments have shown that coated paper could inhibit weed establishment for more than 10 weeks, while uncoated paper was degraded and ineffective after 6–9 weeks. Another approach is surface mulching with harvested plant material, often cereal straw or cuttings of legumes (Fig. 1) produced in an adjacent field. However, experimental results and practical experience with cuttings of legumes have shown that this practice requires a great area of land. The exact rate of mulch biomass needed for efficient weed control depends on several factors such as weed species composition, weed developmental stage at mulching, the rate of mulch decomposition and the competitiveness of the crop, which again depends on species/cultivar, fertilization, etc. Because of the many influencing factors, it is difficult to estimate the amount of plant cuttings or straw needed, but many studies have shown that this should exceed 500 g (DW) m⁻². One way of



Fig. 1 Cuttings of white clover used as mulch in white cabbage. The use of cuttings as mulch in vegetables influences several growth aspects, e.g., effects on weeds and the predator fauna (photo: Lars Olav Brandsæter).

decreasing the need for external biomass can be to mulch only in-row and to hoe between-row. Many mulches may not have a satisfactory effect on perennial weed species, and such weed species should be removed before establishing the mulch.

As already mentioned, some organic mulches are also used for arthropod pest control. In carrots, sawdust or legume clippings have been tested for control of carrot psyllid (*Trioza apicalis*) (Fig. 2). The damage by this herbivore was significantly reduced and yield significantly increased by the application of these materials. Studies in carrot fields have also indicated large differences in predator fauna between unmulched and mulched plots. For example, the carbid beetle, *Bempiidion lampros* (Herbst), was caught in lower numbers in grass-mulched plots. On the other hand, Staphylinid beetles were found to be more numerous in mulched plots. Other studies have shown that reproduction of phytoparasitic nematodes, e.g., the root-knot nematode (*Meloidogyne incognita*), on tomatoes differs between plastic films of different colors. Furthermore, the use of reflective mulches in squash have delayed mosaic virus epidemics due to a reduction in the aphid vector populations. Many investigations have concluded that different herbivore populations are significantly lower in plots with various colored plastic (or other materials) mulches, and it has been proposed that mulches, under high insect stress, should be selected for their effects on insects in addition to effects on yield directly.

Orchards

Mulching can be an advantageous practice in fruit production, especially in organic farming. Similar to vegetable production, black plastic has also been found to block weeds effectively in orchards. To increase durability, woven plastic can be used. Bark, wood chips, or a compact layer of straw is also a suitable mulch material, but a layer of minimum 10–15 cm is

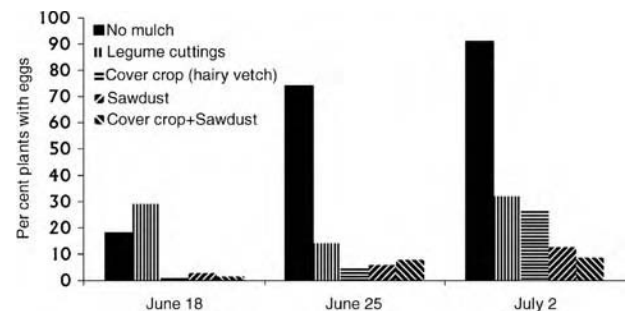


Fig. 2 Example of the effect of mulches and cover crop in carrots for carrot psyllid management (graph: Richard Meadow, Norwegian Crop Research Institute).

needed for long-term weed effects. Decomposing material like bark, which is often rich in nutrient elements, has been found to have an insufficient effect in weed control. Mulching experiments in fruit production have also shown impacts on other pests, e.g., reduced populations of *Pythium ultimum*, in the upper root zone when using black plastic mulch. Black plastic mulch is also reported to reduce densities of several phytoparasitic nematodes in the upper root zone. The effects in the upper root zone are probably due to temperature.

Although mulching can be an interesting practice in fruit production, there are some disadvantages. There is a problem with managing weeds at the margins of mulched strips, especially when using plastic films. Another problem is weeds growing around the trunks, which have to be removed by hand. Voles also represent a concern when using mulches in orchards, since dense groundcovers provide an ideal vole habitat. The vole problem seems to be correlated to the type of mulch, since tree damage has in some studies been observed less frequently under wood chips than under other mulches. As mentioned earlier, problems with soil-borne diseases can be reduced when using mulch; however, experiments have shown that some diseases can also be enhanced, e.g., *Phytophthora*, a root disease found when using straw mulch.

ECONOMICAL AND TECHNICAL CONSIDERATIONS

Machinery is available for laying plastic or paper and punching holes for transplanting. Problems encountered with the use of these mulches include application, tearing, application of fertilizer, overhead irrigation, and disposal after harvest. Natural mulches have beneficial effects such as adding organic matter and nutrients to the soil and preventing water loss. However, they are difficult and time-consuming to apply, and in some cases, they may introduce weed seeds.

Mulching practice, for example in orchard production, is often much more expensive than the use of herbicides. However, herbicides are not accepted in organic farming, so mulching would therefore be a more beneficial practice in pesticide-free systems. Because most mulches are more expensive to establish and maintain compared to herbicides, it must be an important requirement that the benefits of mulches compensate for their additional expense. Economic studies have indicated, however, that for some cropping systems, e.g., orchard production, the increased crop value in mulched systems justifies the greater costs.

BIBLIOGRAPHY

- Dale Monks, C.; Monks, D.W.; Basden, T.; Selders, A.; Poland, S.; Rayburn, E. Soil temperature, soil moisture, weed control, and tomato (*Lycopersicon esculentum*) response to mulching. *Weed Technol.* **1997**, *11*, 561–566.
- Duncan, R.A.; Stapleton, J.J.; McKenry, M.V. Establishment of orchards with black polyethylene film. Mulching: effect on nematode and fungal pathogens, water conservation, and tree growth. *Suppl. J. Nematol.* **1992**, *24* (4S), 681–687.
- Egley, G.H. Stimulation of weed seed germination in soil. *Rev. Weed Sci.* **1986**, *2*, 67–89.
- Hembry, J.K.; Davies, J.S. Using mulches for weed control and preventing leaching of nitrogen fertiliser. *Acta Hort.* **1994**, *371*, 311–316.
- Rämert, B. The influence of intercropping and mulches on the occurrence of polyphagous predators in carrot fields in relation to carrot fly (*Psila rosae* (F.)) (Dipt., Psilidae) damage. *J. Appl. Entomol.* **1996**, *120*, 39–46.
- Riley, H.; Brandsäter, L.O. The use of living or surface mulch to supply nutrients and control weeds in organic vegetable growing. *Acta Hort.* **2000**, *563*, 171–178.
- Ruberson, J.R. *Handbook of Pest Management*; Marcel Dekker: New York, 1999.
- Schonbeck, M.W. Weed suppression and labor costs associated with organic, plastic, and paper mulches in small-scale vegetable production. *J. Sustainable Agric.* **1998**, *13* (2), 13–33.

Multilateral Environmental Agreements

Ulrich Schlottmann

Head of Division IG II 3, Federal Ministry for the Environment, and Nature Conservation and Nuclear Safety, Bonn, Germany

Mirco Kreibich

Division 402: Regional Development Banks, IFAD, Federal Ministry for Economic Cooperation and Development, Bonn, Germany

INTRODUCTION

The emergence of modern pest management methods in the last century has inevitably caused damage to natural resources and a loss of biodiversity. In particular, the widespread use of agrochemicals in both the developed world and the developing world is posing risks to human health and the environment. In light of these problems, several multilateral environmental agreements (MEAs) on chemicals management and the protection of biodiversity were developed within the framework of the United Nations Environment Programme (UNEP) and the United Nations Food and Agricultural Organization (FAO). This article gives an overview of the most important treaties with regard to pest management.

PEST MANAGEMENT AND THE ENVIRONMENT

According to the Organization of Economic Cooperation and Development (OECD), the chemical industry had an annual revenue of US\$1500 billion in 1998. The OECD further predicts an annual output of US\$2360 billion (in 1996 prices) by the year 2010, with most of this increase in non-OECD countries.^[1] Although fertilizers and pesticides make up only US\$90 billion (7%) of the total output, their open application in nature severely enhances the dangers.^[1]

Obsolete pesticides, chemical accidents, acute poisoning, and pesticide residues in food and the environment are serious problems, especially in developing countries, and are a threat to the global environment. The FAO calculated that up to 500,000 tons of obsolete pesticides are stocked in non-OECD countries.^[2] Approximately 20% of these stocks consist of persistent organic pollutants (POPs). Conditions of obsolete stocks range from extremely good to toxics leaking from containers into the surroundings. UNEP Chemicals identified country-specific problems in all countries investigated in a number of case studies, ranging from toxic pesticide residues in groundwaters in

Burkina Faso to levels of dichlorodiphenyltrichloroethane (DDT) and other POPs above the permissible levels in Vietnamese food, at least partly caused by the use of pesticides.^[3] According to the World Health Organization (WHO) and the UNEP, there are about 1,000,000 cases of acute accidental pesticide poisoning every year, 20,000 of which end lethally.^[4]

The more recent development of living modified organisms (LMOs) as a pest management technology also gives rise to concern. Without doubt, there is a great potential for the use of biotechnology in agriculture with possible positive effects for the environment. But science is still uncertain about environmental and health-related risks and the socioeconomic effects of a widespread use of LMOs. Critics argue that the use of LMOs would reduce the genetic diversity of crops, increase farmers' dependence on large seed-producing companies, may have devastating ecological effects, does not necessarily lead to a decrease in the use of agrochemicals, and does not serve as a tool to reduce famine in developing countries.^[5,6]

DEVELOPMENT OF RELEVANT INTERNATIONAL ENVIRONMENTAL LAW

A multilateral context to tackle these problems is necessary as the problems are international and transboundary in nature, and therefore require international regulatory frameworks. The 1992 Rio UN Conference on Environment and Development (UNCED) devoted several chapters to pest management-related issues in its Agenda 21, including chapters on planning and management of land resources, sustainable agriculture and rural development, conservation of biodiversity, management of biotechnology, management and use of water resources, management of toxic chemicals, and management of hazardous wastes.^[7]

The first of these MEAs was developed in the 1980s, but the follow-up process to the Rio Conference in particular saw the adoption of several chemicals and biodiversity-related MEAs. Now there exists a wide

Table 1 Status of multilateral environmental agreements in chemical safety and the protection of biodiversity

| Name of MEA | Year of adoption | Status of signatures | Status of ratifications | Entry into force |
|--|------------------|----------------------|-------------------------|------------------|
| Basel Convention on the Control of Transboundary Movements of Hazardous Wastes and Their Disposal | 1989 | 151 + EC* | 154 + EC* | 1992 |
| United Nations Convention on Biological Diversity (UNCBD) | 1992 | 167 + EC* | 186 + EC* | 1993 |
| Aarhus Protocol on Persistent Organic Pollutants (POPs) to the UN-ECE Convention on Long-Range Transboundary Air Pollution | 1998 | 35 + EC* | 14 | Not yet |
| Rotterdam Convention on the Prior Informed Consent for Certain Hazardous Chemicals in International Trade | 1998 | 72 + EC* | 40 + EC* | Not yet |
| Cartagena Protocol on Biosafety to the UNCBD | 2000 | 102 + EC* | 44 + EC* | Not yet |
| Stockholm Convention on Persistent Organic Pollutants | 2001 | 150 + EC* | 30 | Not yet |

EC = European Community.

As a so-called "regional economic integration organization," the EC is entitled to sign and to ratify the conventions, in addition to its individual member states. The EC does not have an additional vote in the Conferences of Parties, however.

range of legally binding instruments which are directly or indirectly dealing with the issue of pest management. Table 1 and the following sections give an overview of the most important MEAs in the area.

THE BASEL CONVENTION

The Basel Convention's objective is to regulate transboundary movements of the approximately 400 million tons of hazardous wastes that are produced annually. These wastes may be hazardous for their toxic, poisonous, explosive, corrosive, flammable, ecotoxic, or infectious characteristics. In particular, the prevention of illegal traffic and the environmentally sound management and disposal of these wastes are the convention's major aims. To this end, the convention has established technical guidelines for the management of numerous different types of waste. Recently, an amendment to the convention has banned the export of hazardous wastes to developing countries even if they are not parties to the convention ("Basel Ban").

The convention is of importance for pest management in two respects. Firstly, it restricts the export and the import of stockpiles of obsolete pesticides and pesticide wastes. Secondly, it gives guidance on how to manage obsolete pesticides including the prevention, the minimization, and the recycling of pesticide wastes. In addition, its regulations have direct links to other relevant conventions, such as the Rotterdam Convention or the Stockholm Convention.

THE BIODIVERSITY CONVENTION AND THE CARTAGENA PROTOCOL

The UN Convention on Biodiversity (UNCBD) aims at conserving the global biological diversity and its sustainable use, and at ensuring a fair and equitable share of benefits arising from the commercial use of genetic resources. To this end, the convention contains a number of provisions for parties, such as the establishment of protected areas, the promotion of the protection of ecosystems, and the regulation of access to genetic resources. The national implementation of the convention's goals has a particular impact on forestry, agriculture, and fisheries, and thus on pest management (e.g., by encouraging governments to use their national resources in a sustainable manner, or by preventing the introduction of alien species that could threaten ecosystems, habitats, or species). Raising crops within mixed ecosystems or integrated pest management is promoted in order to minimize pesticide use.

The Cartagena Protocol regulates the safe management of transboundary movements of LMOs and was established as a protocol to the UNCBD. Similar to the PIC procedure of the Rotterdam Convention (see section "The Rotterdam Convention"), it establishes a so-called advance informed agreement (AIA) procedure to ensure that countries obtain the information necessary to make an informed decision on whether or not to allow the import of LMOs. Commodities that may contain LMOs are to be clearly labeled when exported.

THE ROTTERDAM CONVENTION

The Rotterdam Convention builds upon a voluntary procedure established and operated by the UNEP and the FAO (i.e., FAO's "Code of Conduct on the Distribution and Use of Pesticides" of 1981 and particularly UNEP's "London Guidelines for the Exchange of Information for Chemicals in International Trade" of 1987). The convention's objective is to promote an information exchange on the characteristics of certain hazardous chemicals among trading countries with the aim of enhancing cooperation. The chemicals also include a list of pesticides (initially 22 pesticides, but new pesticides are being added on a regular basis). A chemical listed in the convention's annexes can be exported only with the prior informed consent (PIC procedure) of the importing party. Importing parties are therefore given the power to decide whether or not they wish the import of certain hazardous substances.

The PIC procedure was addressed primarily to developing countries. The unregulated import particularly of pesticides has left these countries with a burden of obsolete pesticide stocks, wastes, and severely hazardous chemicals on their domestic markets, combined with a lack of knowledge about the chemicals' properties and a lack of authorities dealing with the risks of chemicals.

THE STOCKHOLM CONVENTION AND THE UN-ECE POP PROTOCOL

The Stockholm Convention prohibits or severely restricts the production, use, or release of initially 12 POPs. The aim is to eliminate POPs on a global scale. These chemicals are characterized by their toxicity, persistency, tendency to bioaccumulate, and potential to travel long distances. The substances listed in the annexes of the convention include several pesticides (aldrin, chlordane, dieldrin, DDT, endrin, heptachlor, mirex, and toxaphene), industrial chemicals (hexachlorobenzene and polychlorinated biphenyls), and unintentionally released by-products of production and incineration processes (dioxins and furans). Further measures include regulations concerning wastes and stockpiles, the regulation of trade with these substances, general and specific exceptions, and financial and technical aid. The list of substances is not conclusive. Further POPs will be subject to evaluation according to criteria defined in the convention and are likely to be added.

The convention is of relevance for pest management practitioners because it prohibits the use of several pesticides that have been—and, in some countries, still are—of great importance in pest management. In most

industrialized countries, the use of these substances has long been prohibited. Export, however, to developing countries was still allowed. Developing countries also act as producers themselves, so that some of these substances are still found in great abundance. The major burden of implementing the convention will be on developing countries. But industrialized countries also have a great interest in its implementation: POPs have the ability to travel by air, water, and migratory species from their southern sources toward the poles and thus cause major problems in industrialized countries.

The POP Protocol of 1998 was a regional forerunner to the Stockholm Convention, developed within the UN Economic Commission for Europe (UN-ECE), a group that comprises 55 mostly European and some developed non-European countries, including the United States. Compared to the Stockholm Convention, it contains four additional POPs (chlordecone, hexabromobiphenyl, lindane, and polycyclic aromatic hydrocarbons).

Apart from the abovementioned MEAs, several smaller or regional instruments with certain relevance to the use and management of pesticides were established. These include the Convention Concerning Safety in the Use of Chemicals at Work of 1990 within the framework of the United Nations International Labor Organization (ILO), the European Agreement Concerning the International Carriage of Dangerous Goods by Road of 1957, and the Convention for the Protection of the Marine Environment of the Northeast Atlantic (OSPAR Convention) of 1992.

CONCLUSION

In this article a number of MEAs were presented with direct or indirect relevance to pest management. In the past, pest management techniques and unsustainable agricultural practices have contributed to the degradation of the rural environment. The MEAs negotiated over the last years provide an international legal framework for the global protection of the environment from adverse effects caused by the use and trade of certain pesticides, toxic wastes, living modified organisms, and the loss of biological diversity.

REFERENCES

1. Organisation for Economic Co-operation and Development. *Environmental Outlook for the Chemicals Industry*. OECD: Paris, 2001; 164 pp. reference year 1996.
2. Food and Agricultural Organisation of the United Nations. *Baseline Study on the Problem of Obsolete Pesticide Stocks*. Pesticide Disposal Series 9; FAO: Rome, 2001; 36 pp.

3. United Nations Environment Programme. *Country Case Studies on Persistent Organic Pollutants (POPs)*; UNEP Chemicals and the Inter-organisation Programme for the Sound Management of Chemicals (IOMC): Geneva, 2000; 318 pp.
4. Holzmann, A. Informationsaustausch und PIC-Verfahren (Prior Informed Consent) beim Export und Import von Pflanzenschutz- und Schädlingsbekämpfungsmitteln. *Nachrichtenbl. Pflanzenschutz* **1995**, 47 (7), 181–186. quoting WHO data of the late 1980s.
5. Dürkop, J.; Dubbert, W.; Nöh, I. *Beitrag der Biotechnologie zu Einer Dauerhaft Umweltgerechten Entwicklung*; UBA Texte 1/99; Umweltbundesamt: Berlin, 1998; 205 pp.
6. de Kathen, A. *Transgenic Crops in Developing Countries. A Report on Field Releases, Biosafety Regulations and Environmental Assessment*; UBA Texte 58/99; Umweltbundesamt: Berlin, 1999; 132 pp.
7. United Nations Conference on Environment and Development. *Agenda 21*; United Nations Department of Public Information: New York, 1993; 294 pp.

National Pesticide Poisoning Surveillance

Hans Persson

Margareta Palmborg

Swedish Poisons Information Centre, Stockholm, Sweden

INTRODUCTION

Among pesticides, there are many products with highly toxic ingredients. Exposure to these substances may result in severe poisoning. Pesticide poisoning is a serious health problem which is especially evident in developing countries.^[1] Knowledge about the pattern and severity of pesticide poisonings is necessary for the implementation of an effective prevention program. Studies carried out in the international level, using a standardized protocol, would be the ideal way to obtain a better understanding of the problem globally. However, national surveys on pesticide poisoning may also be very useful in elucidating special aspects on the problem, and in this respect, poisons centers can play an important role.

THE POISONS CENTER

The first poisons information centers, nowadays more often named just *poisons centers*, were established in North America and Europe in the late 1950s and early 1960s. Gradually, units of this kind have become available in all continents, but many developing countries still lack this kind of service. Their main task is to provide information on risks, symptoms, and treatment of poisoning. The principal target group is medical professionals who need guidance in the management of unusual or complicated cases of poisoning. However, in addition, many centers also take calls from the general public, workplaces, etc.

A poisons center, which responds to calls from both medical professionals and the public, will receive inquiries concerning poisoning accidents involving all kinds of products, and the poisons center is faced with a lot of information about the poisoning incident. Access to this information makes the poisons center well suited for performing *toxicovigilance*, including surveys on poisonings that involve particular groups of toxic agents. Therefore, poisons centers could, within the frame of their routine work, design prospective follow-up studies where a number of variables are analyzed: the frequency of a certain type of poisoning (e.g., pesticide poisoning), age and sex distribution of

those poisoned, products involved, circumstances, and clinical course.

A poisons center survey on pesticide poisonings could be conducted in the following way:

- All inquiries to the poisons center are registered in connection with the initial telephone call to the center.
- Information is actively requested concerning the patient (age, sex, occupation), the product(s) involved, circumstances surrounding the incident (e.g., intentional or accidental exposure, amounts and routes of exposure, factors precipitating the accident), and clinical symptoms.
- All information gained is documented on a specially designed protocol.
- Telephone follow-up is made in all cases, except in those where no symptoms whatsoever can be expected (e.g., because of low toxicity of the product, strongly diluted preparations, or minimal exposure).
- Whenever the poison center is contacted by a hospital, or if the center has advised on admission to a hospital, a discharge summary is requested concerning that case.

In some countries, hospitals send discharge summaries to poisons centers routinely. In Sweden, the poisons center is, irrespective of any ongoing study, receives discharge summaries covering a little more than one third of all in-patients treated for poisoning in the country. Thus medical documentation on exposure, symptoms, and outcome for a very large group of patients is kept at the poisons center. These cases constitute an indispensable source of information that can be studied retrospectively and used as a reference material in studies on specific types of poisoning.

OTHER INFORMATION SOURCES

In addition to the information generated within the poisons information service as outlined above, any other relevant information sources should be looked for, ensuring a picture that is as complete as possible. These other sources and their availability may vary from one country to another because of local conditions.

Mortality is always a key parameter in epidemiological studies. As all poisoned patients do not die in hospital, it is necessary to consider national mortality statistics to obtain a reliable idea of actual mortality. In Sweden all deaths, irrespective of their cause, are reported to the National Central Bureau of Statistics. In death certificates, the cause of death is indicated as interpreted by the responsible physician. It is natural to include data from this register in any nationwide study on the morbidity and mortality of a particular poison.

Some pesticide poisoning cases may be treated in occupational medicine clinics, where additionally useful information may be gathered. Such clinics could therefore be approached separately with a request to report on any pesticide poisonings they come across.

In larger industries, there are special occupational health services available. If such units have treated poisoning cases, information about these cases could also be evaluated and included in a survey.

The national authority responsible for approval of pesticides, local authorities handling information on pesticides in the community, and the national authority for industrial welfare are other bodies that keep useful information for the evaluation of pesticide poisoning problem in a country. But, perhaps more importantly, such institutions will benefit greatly from survey results, which may serve as guide in regulatory and preventive work.

RESULTS FROM TWO STUDIES IN SWEDEN 1984 AND 1994

Two prospective studies of pesticide poisoning in Sweden were performed by the Swedish Poisons Information Centre during the years 1984 and 1994 respectively.^[2] They could serve as an illustration of how the principles outlined above have been applied in practice.

The incidence of acute pesticide poisoning in Sweden is low. The number of inquiries concerning pesticides increased during the 10-year interval between the two studies (from 885 to 1703); during that period, the total number of inquiries to the poisons center almost doubled, too. Therefore, the proportion of pesticide-related inquiries remains constant around 3%.

The number of inquiries to the poisons center does not, however, reflect the real number of human exposures. If the substantial number of calls related to animal exposures and questions of a more general nature are deducted, we are left with 493 human poisoning cases in 1984 and 774 in 1994. Most of these were treated as outpatients or at home. Based on obtained hospital discharge summaries, approximately 50 to 60 patients are annually treated as in-patients because of pesticide poisoning in Swedish hospitals.

Most accidents occur at home, where an increase in incidence is observed. On the other hand, the number of occupational accidents with pesticides has decreased between the two study years. Within the respective materials, the proportion of occupational accidents decreased from 30% to 9%.

In both studies, children were figured in most accidents (65%) and males dominated slightly (60%). The route of exposure differs between adults and children. Referring to the data collected in 1994, ingestion is the dominant route of exposure among children, whereas inhalation is more common in adults, followed by skin contact, ingestion, eye exposure, and a combination of several routes.

Insecticides constitute the largest group among poisoning agents both in 1984 and 1994, with organophosphorus compounds and carbamates topping the list (Table 1). Most of the inquiries concern pesticides with low toxicity, and there is a notable increase since 1984 for less dangerous products. This may be related to the introduction, between the study years, of two new groups of low toxicity pesticides, borax and

Table 1 Pesticides involved in two studies performed in Sweden

| Pesticide | Cases 1984 | Cases 1994 |
|--|------------|------------|
| Insecticides | 162 | 349 |
| Borax | 4 | 94 |
| Organochlorines | 1 | 1 |
| Organophosphates, carbamates | 98 | 106 |
| Pyrethrins, rotenone | 54 | 22 |
| Pyrethroides | 5 | 126 |
| Rodenticides | 52 | 117 |
| Crimidine | 1 | — |
| Superwarfarins | — | 39 |
| Warfarin | 49 | 58 |
| Other/Unknown | 2 | 20 |
| Repellents | 74 | 76 |
| Herbicides | 41 | 64 |
| Dinoseb | 1 | — |
| Diquat | 1 | — |
| Phenoxy acids | 22 | 17 |
| Glyphosate | 8 | 39 |
| Ferrous sulfate | — | 7 |
| Sodium chlorate | 9 | 1 |
| Antifouling products and wood preservatives | 61 | 66 |
| Fungicides | 2 | 3 |
| Other/unknown/unspecified | 101 | 99 |
| All pesticides | 493 | 774 |

pyrethroides. Rodenticides are the second largest group in 1994, and there is a twofold increase over the 10-year period. This change relates to the great number of accidents with long-acting anticoagulant rodenticides—the superwarfarins, a fairly new group of rodenticides. Acute unintentional ingestion of these substances will impose a special problem, especially among children, as the risk of poisoning cannot be excluded without medical examination, including laboratory tests. Among herbicides, accidents with glyphosate have become frequent since 1994.

The surveys in 1984 and 1994 have shown that in most cases, symptoms were either lacking or mild. In 1994, only 3% of the cases were classified as “moderate” according to the Poisoning Severity Score (PSS)^[3] and in 1984 symptoms of “moderate” poisoning developed in 4% of the cases. Only one poisoning case was classified as “severe” during each of the surveys.

One fatal case occurred in the 1994 study. Fatal outcome in pesticide poisoning is rare in Sweden. To assess the situation as regards mortality over a longer period, all deaths resulting from pesticide exposure as reported to the National Central Bureau of Statistics from 1969 to 1994 were studied. There was a total of 21 fatal cases during this 26-year period, but only three of them occurred as a result of accidental poisoning. The remaining 18 cases were all of suicidal or unclear origin. Organophosphorus pesticides or carbamates were involved in about half of the lethal cases.

A FAVORABLE SITUATION

The incidence of acute poisoning with pesticides in Sweden is low, and extremely low compared with the overall global situation.^[1] Accidental exposures at home dominate. Poisoning incidents caused by exposure at work is decreasing. Self-poisonings are uncommon and accidents rarely result in significant poisoning. Fatal outcome has been exceptional during recent years and has almost invariably been the result of a suicidal act.

The favorable situation in Sweden may probably be ascribed to a number of interacting factors. Because of climate conditions, there is a limited need of highly toxic pesticides. Sweden is also striving to diminish the overall use of pesticides.^[4] A strict legislation of pesticides in the country has resulted in withdrawal of the most toxic substances, limited use of other toxic pesticides, and restricted availability of certain pesticides to the general public. Training courses are compulsory for workers using pesticides occupationally. Finally, pesticides are not traditionally used in suicidal poisoning.

CONCLUSIONS

There are advantages in performing surveys on selected groups of toxic agents within the routine work of a poisons center. The study can easily be performed among other activities in the center, and the information gained is difficult to retrieve elsewhere. A standardized telephone follow-up close to a primary contact because of an incident provides information on many parameters: frequency, circumstances, age and sex distribution, routes of exposure, products involved, and symptoms. If the poisons center responds to inquiries from a defined population and area, this is an advantage when comparisons are made between surveys performed during different periods. Product information about pesticides and documentation on the toxicity of the ingredients in specific products is available in the center. This will make it possible to identify the substances involved and will help in evaluating the risk of poisoning in a certain case. Documentation and experience in the center will make it possible to judge on the relevance of symptoms described in a particular case. Assessment of the severity of poisoning is fundamental for the proper understanding of the problem as a whole—figures on frequency alone are insufficient. This qualitative aspect of the problem will be provided for by using a classification scheme for the severity of poisoning, e.g., the “PSS.”^[3] Admittedly, poisons center studies have also their limitations. For instance, all poisoning incidents are not known by the center because there are cases when it is not contacted at all. Furthermore, some inquiries are impossible to follow up for various reasons.

In spite of certain limitations, information obtained in a poisons center survey may be a most useful tool in assessing the pattern and severity of pesticide poisoning in a country. This, in its turn, may provide guidance for authorities on how to reduce morbidity and mortality of poisoning from pesticides.

REFERENCES

1. WHO/UNEP. *Public Health Impact of Pesticides Used in Agriculture*; World Health Organisation: Geneva, 1990.
2. Persson, H.; Palmberg, M.; Irestedt, B.; Westberg, U. Pesticide poisoning in Sweden—Actual situation and changes over a 10 year period. *Przegl. Lek.* **1997**, *54* (10), 657–661.
3. Persson, H.; Sjöberg, G.; Haines, J.; Pronczuk de Garbino, J. Poisoning Severity Score (PSS). Grading of acute poisoning. *J. Toxicol., Clin. Toxicol.* **1998**, *36* (3), 205–213.
4. Ekström, G.; Hemming, H.; Palmberg, M. Swedish pesticide risk reduction 1981–1995: Food residues, health hazard, and reported poisonings. *Rev. Environ. Contam. Toxicol.* **1996**, *147*, 119–147.

Natural Enemies and Biocontrol: Artificial Diets for Rearing

Simon Grenier

UMR INRA/INSA de Lyon, Biologie Fonctionnelle Insectes et Interactions (BF21),
Villeurbanne Cedex, France

Patrick De Clerq

Laboratory of Agrozoology, Department of Crop Protection, Faculty of Agricultural and
Applied Biological Sciences, Ghent University, Ghent, Belgium

INTRODUCTION

Arthropod parasitoids and predators used in biological control strategies are at present mainly produced on natural or alternative hosts or prey. However, their large-scale production may be more convenient and cost-effective when using artificial diets/media. Studies aiming at the successful development of arthropod parasitoids and predators under artificial conditions have started a long time ago, but the practical use of insects and mites grown on artificial diets is still in its infancy. Besides their use for the production of natural enemies, artificial media may be valuable tools for physiological and behavioral studies of entomophagous arthropods due to a simplification of their environment. Different types of artificial diets with or without insect additives can support the development and/or reproduction of natural enemies. Successes have been achieved for several species of parasitoids and predators but these have mainly been restricted to an experimental level. Comparisons of the performances of artificially vs. naturally reared natural enemies (as quality control) have primarily been conducted in the laboratory, and only very rarely in the field. The promising results achieved in recent years open up new prospects for natural enemy producers.

ARTIFICIAL DIETS FOR PREDATORS AND PARASITIDS

The culture of entomophagous insects and mites involves rearing not only of the host/prey, but often also of the host's/prey's plant food, and thus requires a tritrophic level system. Different steps were taken to try to reduce the production line for entomophagous arthropods. The complete line comprises plant growing, host/prey rearing, and parasitoid/predator rearing. The simplified line includes the use of artificial diets instead of plants for the phytophagous host/prey, or of factitious hosts/prey that are easier to rear in the laboratory than the natural food (e.g., eggs of *Ephesia*

kuehniella or *Sitotroga cerealella*, larvae of *Galleria mellonella* or *Tenebrio molitor*). The ultimate reduction of the production line consists only of an artificial diet for direct parasitoid/predator rearing. Mass rearing entomophagous insects on artificial media, first suggested 60 years ago, holds the promise to increase the ease and flexibility of insect production, including automation of procedures, and to reduce cost. The early and subsequent efforts at developing artificial diets have extensively been reviewed.^[1-3] The basic qualitative nutritional requirements of parasitoids and predators are similar to those of free-living insects. But the very fast growth of some parasitoids such as tachinid larvae requires a perfectly well-balanced diet^[4] to minimize intermediate metabolism and toxic waste product accumulation.

Essentially, two types of artificial diets can be distinguished: Those including and those excluding insect components. The availability of media without insect components offers a greater independence from insect hosts/prey, even if in some countries insect components are cheap and easily available by-products, e.g., from silk production in Asia or South America.^[5] In diets containing insect additives, such varied components as hemolymph, body tissue extract, bee brood extract or powder, egg juice, or homogenate of the natural host have been used. Products of insect cell culture have also been incorporated into diets as host factors. The composition of most media for in vitro rearing of *Trichogramma* egg parasitoids is based on lepidopterous hemolymph.^[6] Media for the tachinid fly *Exorista larvarum*, the chalcid wasp *Brachymeria intermedia*, and the ichneumonid wasp *Diapetimorpha introita* contain various insect components. Bee extracts or bee brood have been commonly added in diets for predatory coccinellids.^[1,5] Only few diets devoid of insect additives are composed of ingredients that are fully chemically defined in their composition and structure. Besides proteins or protein hydrolysates, most of such diets contain crude or complex components, e.g., hen's egg yolk, chicken embryo extract,

calf serum, cow's milk, yeast extract or hydrolysate, meat or liver extract, or plant oils. Beef or pork meat and liver have extensively been used as basic components of diets for feeding coccinellids and several predatory heteropterans.^[1,5]

SUCCESSSES AND FAILURES WITH ARTIFICIAL DIETS

Both biochemical and physical aspects determine the success of an artificial diet. Artificial diets should be nutritionally adequate to support development and reproduction of an insect and should be formulated in such a manner that the medium is easily recognized and accepted for feeding or oviposition; the food should be readily ingested, digested, and absorbed.^[7] For parasitoids, the diet must also allow the growing larvae to satisfy other physiological needs like respiration and excretion without diet spoiling. The best results on artificial media were obtained with idiobiontic parasitoids such as egg or pupal parasitoids and with polyphagous predators. Different tachinid species were also successfully grown in vitro, but the koinobiontic Hymenoptera appear the most difficult group to be reared in vitro, probably because of a close relationship with their living host that supplies them with crucial growth factors. Ectoparasitoids are generally easier to culture in vitro than endoparasitoids for which the diet is also the living environment of the immature stages.^[2] Several predatory insects have been reared for successive generations on artificial diets, including heteropterans (e.g., *Geocoris punctipes*, *Orius laevigatus*, *Podisus maculiventris*), coccinellids (e.g., *Coleomegilla maculata*, *Harmonia axyridis*), and chrysopids (e.g., *Chrysoperla carnea*, *Chrysoperla rufilabris*).^[3]

Artificial rearing of natural enemies has mostly remained at an experimental level, and the practical experience with natural enemies produced in artificial conditions has remained quite limited. Wasps of the genus *Trichogramma* reared on factitious host eggs are the most common agents used worldwide in biological control in many field crops and forests. In China, *Trichogramma* spp. and *Anastatus* spp. produced on a large scale in artificial host eggs have been released on thousands of hectares of different crops with a parasitization rate above 80%, leading to an effective pest control level equal to that of naturally reared parasitoids.^[5] In the U.S.A., field tests with encouraging first results were conducted using the pteromalid parasitoid *Catolaccus grandis* reared for successive generations on artificial diet for the control of the cotton boll weevil *Anthonomus grandis*.^[5] Since the late 1990s, biocontrol companies in the U.S.A. and

Europe have started producing a number of natural enemies (partially) on artificial diets.

QUALITY CONTROL OF NATURAL ENEMIES PRODUCED ON ARTIFICIAL DIETS

Long-term rearing on artificial diets could lead to genetic bottleneck effects inducing high selection pressure on the entomophages and possible reduction of their effectiveness. Periodic population renewals from nature may circumvent this drawback. The use of natural enemies in augmentative biological control requires a reliable mass production of good quality insects. Therefore, quality control is a key element for the efficiency and the long-term viability of biological control. The quality control procedures developed for in vivo production of entomophages could be recommended as a first approach for in vitro production.^[8] Many parameters can be used as quality criteria. Size, weight, life cycle duration, survival rate, and especially fecundity, longevity, and predation/parasitization efficiency are the most relevant characters.^[5] Besides its value as a quality criterion, the biochemical composition (based upon carcass analyses) of the insects produced on artificial diets may be a powerful tool for improving the composition and performance of the diets through the detection of excess or deficiency of some nutrients. Often, different criteria are closely linked; hence, the quality control process may be simplified if one easily measured parameter can be used to predict another one that is more complex or time consuming to determine (e.g., fecundity). Arguably, excellent field performance of the artificially produced natural enemy against the target pest remains the ultimate quality criterion. However, quality assessments of artificially reared natural enemies have mostly been performed at a laboratory scale or in semifield conditions, and only rarely so in practical field conditions.

CONCLUSIONS

At present, rearing systems using natural or factitious foods remain the only effective way for industrial production of most entomophagous insects and mites. However, success achieved for a restricted number of species of parasitoids (e.g., *Trichogramma* spp., *Exorista larvarum*, *Catolaccus grandis*) and predators (e.g., *Orius* spp., *Geocoris punctipes*, *Chrysoperla* spp., *Harmonia axyridis*) has prompted producers to increasingly incorporate artificial diets into their mass rearing systems. Further behavioral and physiological investigations may lead to significant improvements in artificial rearing through a better knowledge of the host-parasitoid and predator-prey relationships.

Besides an easier mechanization of the production line, the use of artificial diets opens new possibilities for preimaginal conditioning of parasitoids/predators to targeted hosts/prey by adding specific chemicals in their food. Artificial diets also seem the only way of mass rearing for some middle-sized egg parasitoids (Encyrtidae, Eulophidae, Eupelmidae, Scelionidae to name a few) that are promising pest control agents but are unable to develop normally in the small lepidopteran substitution host eggs commonly used nowadays (*Ephestia kuehniella*, *Sitotroga cerealella*).

REFERENCES

1. Thompson, S.N. Nutrition and culture of entomophagous insects. *Annu. Rev. Entomol.* **1999**, *44*, 561–592.
2. Grenier, S.; Greany, P.D.; Cohen, A.C. Potential for mass release of insect parasitoids and predators through development of artificial culture techniques. In *Pest Management in the Subtropics: Biological Control—A Florida Perspective*; Rosen, D., Bennett, F.D., Capinera, J.L., Eds.; Intercept: Andover, U.K., 1994; 181–205.
3. Thompson, S.N.; Hagen, K.S. Nutrition of entomophagous insects and other arthropods. In *Handbook of Biological Control: Principles and Applications*; Bellows, T.S., Fisher, T.W., Eds.; Academic Press: San Diego, CA, 1999; 594–652.
4. Grenier, S.; Delobel, B.; Bonnot, G. Physiological interactions between endoparasitic insects and their hosts—Physiological considerations of importance to the success of in vitro culture: an overview. *J. Insect Physiol.* **1986**, *32* (4), 403–408.
5. Grenier, S.; De Clercq, P. Comparison of artificially vs. naturally reared natural enemies and their potential for use in biological control. In *Quality Control and Production of Biological Control Agents: Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI Publishing: Wallingford, U.K., 2003; 115–131.
6. Grenier, S. Rearing of *Trichogramma* and other egg parasitoids on artificial diets. In *Biological Control with Egg Parasitoids*; Wajnberg, E., Hassan, S.A., Eds.; CAB International: Wallingford, U.K., 1994; 73–92.
7. Cohen, A.C. *Insect Diets—Science and Technology*; CRC Press: Boca Raton, U.S.A., 2003.
8. van Lenteren, J.C.; Hale, A.; Klapwijk, J.N.; van Schelt, J.; Steinberg, S. Guidelines for quality control of commercially produced natural enemies. In *Quality Control and Production of Biological Control Agents: Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI Publishing: Wallingford, U.K., 2003; 265–303.

Natural Enemies and Biocontrol: Function in Mixed Cropping Systems

Tibor Bukovinszky
Joop C. van Lenteren

Laboratory of Entomology, Department of Plant Sciences, Wageningen University and Research Centre, Wageningen, The Netherlands

L.E.M. Vet

Department of Multitrophic Interactions, Netherlands Institute of Ecology (NIOO-KNAW), Heteren, The Netherlands

INTRODUCTION

Many natural ecosystems are characterized by high floral and faunal diversity, where variable defenses protect plants from excessive damage, i.e., through a combination of bottom-up and top-down forces acting on herbivores.^[1,2] Compared with monotypic stands, mixed cropping systems (MCS) often have lower pest populations on the crop species. Several hypotheses have been formulated to explain this phenomenon.^[3] The “host-plant-quality” and “resource-concentration” hypotheses and the “appropriate–inappropriate” landing theories describe the negative bottom-up effects of MCS on the development, host plant finding, and acceptance behavior of herbivores.^[4,5] The “enemies hypothesis” (EH) explains the lower pest numbers in MCS by an increased success of natural enemies,^[6] which is a compounded effect of several factors: 1) Diverse habitats offer important resources for natural enemies, such as nectar and pollen, which can be less available in monocultures; 2) generalist natural enemy populations are less likely to fluctuate because of the greater diversity of host or prey species available within the complex environment; and 3) specialist natural enemies show low population fluctuations, because the refuges provided by a complex environment enables their host or prey to escape widespread annihilation.

Predictions of the EH have rarely been supported by conclusive experimental data, and the effect of habitat diversification on parasitism showed results varying from negative to neutral to positive.^[7,8] One reason for these contradictory results is that the EH did not consider that plants in the habitat do not represent a neutral medium in which organisms interact, but that they also convey infochemicals that influence multitrophic interactions.^[7,9,10] Additionally, in most of the studies testing the EH, data were collected of parasitism rates, while host densities were not standardized, thus confounding the effects of MCS with the functional responses of natural enemies. Consequently,

predictions of the EH are based on population responses of natural enemies, while the underlying behavioral mechanisms remain poorly understood. The objective of this paper is to give an overview of how important characteristics of MCS (e.g., greater amount of resource subsidies, higher structural and infochemical complexity, and indirect interactions between natural enemies) influence the functioning of natural enemies and herbivore–natural enemy interactions (Fig. 1).

DIVERSITY OF SUBSIDIES, COMPLEXITY OF INTERACTIONS

Mixed Cropping Systems and Resource Subsidies

Mixed cropping systems often harbor a greater diversity of herbivores that may be exploited by generalist natural enemies in times of prey/host scarcity.^[5] Also, in MCS, the success of natural enemies can be enhanced by the presence of more resource subsidies (i.e., food, alternative prey/host), which increases natural enemy longevity and fecundity.^[5,11] Such resource subsidies may originate from plants directly (e.g., pollen, floral and extrafloral nectar), or indirectly (e.g., honeydew).^[12] Upon receiving a feeding reward, insect parasitoids may concentrate their searching effort for hosts in the vicinity of the food source,^[12] which may improve their functioning in adjacent crops. Hunger level can influence the responses of natural enemies to stimuli associated with their victims or with the food. Thus, in habitats low in sugar sources, there may be a tradeoff for insect parasitoids between searching for hosts and searching for food.

Nevertheless, not all sugar sources contribute equally to the functioning and survival of natural enemies. Floral nectars may vary in their suitability and accessibility for enemies and herbivores.^[13]

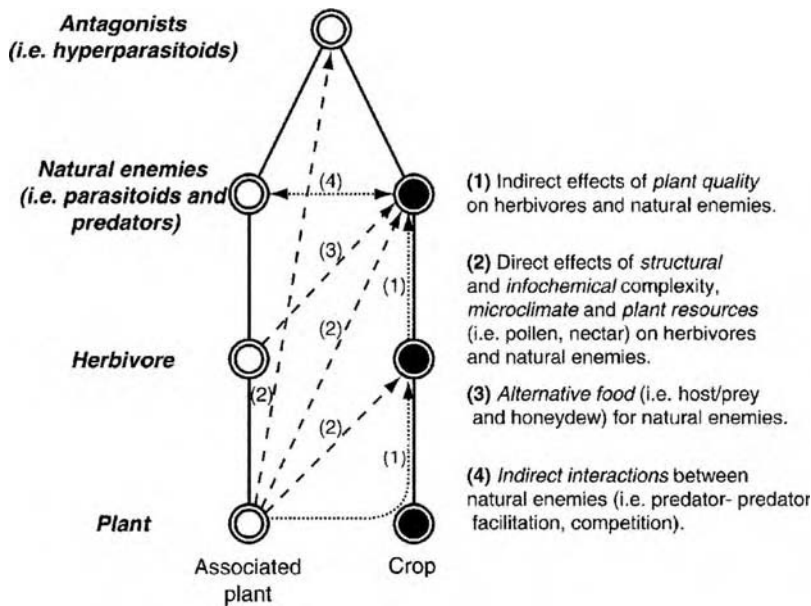


Fig. 1 Possible effects of habitat diversification on organisms in a hypothetical, simplified food web that could influence pest populations in the crop (as described in the text). Circles are trophic levels associated with the crop (black) and the associated plants (white). Solid lines are trophic relationships. Arrows represent direct (dashed) and indirect (dotted) interactions within and between trophic levels.

Inappropriate subsidy of resources may increase pest pressure, either by disrupting the activity of natural enemies or by benefiting the antagonists of natural enemies (i.e., hyperparasitoids) or the pest itself.^[14,15] For these reasons, MCS should provide food for natural enemies in a selective way, which requires in-depth knowledge of the biology of the species in the ecosystem.

that the prey found unoccupied patches, the average probability that predators found the prey, and the stochasticity of individual colonization events.^[23] The essential process allowing persistence of the population was isolation by distance, where subdivision of habitats provided refuges for the prey relative to the predator.

Vegetation Structure

Mixed cropping systems often have a more complex vegetation surface and a higher leaf area index than monotypic stands. As a result, MCS can provide shady, more humid microclimates, leading to higher survival and larger population levels of natural enemies.^[11,16] Structural complexity of the environment may influence the mobility of natural enemies and thus their foraging success. For example, a change in surface area, connectivity, or complexity of plant surface can affect prey/host encounters and thus herbivore-carnivore dynamics.^[17,18] Huffaker's classical studies showed that highly complex environments increased the persistence of predator-prey systems,^[19,20] leading to the prediction that complexity "stabilizes" predator-prey dynamics. However, a study on an aphid-ladybird system found a destabilizing effect of spatial heterogeneity on predator-prey dynamics, leading to prey outbreaks,^[21] where the outcome of the dynamics depended on the searching behavior of the predatory beetles and on the distribution of their prey.^[22] Ellner et al.'s^[23] study established that the greater persistence of a predator-prey system in spatially heterogeneous environments resulted from the average probability

Volatile Information

Plant-derived infochemicals are often used by natural enemies in the process of host/prey location.^[10,24] Vegetation diversity also increases the complexity of infochemical cues. Therefore, the efficient use of specific plant cues by insect parasitoids in a habitat with many non-infested or non-host infested plant species may be hampered, because of the high "background noise." Olfactory disruption by odor masking has been suggested for natural enemies,^[25] but it has rarely been tested (but see Ref.^[26]). Data suggest that the efficiency of a natural enemy that uses plant volatiles in prey/host finding depends on its capability to discriminate between "signal" (i.e., host-infested plants) and "background noise" (i.e., non-infested and non-host infested plants).^[3,26-28] For example, it can be very difficult for the natural enemy to find the host in a monoculture, if the host-infested plant does not give specific information compared to non-infested plants. If infested plants give specific signals, the searching efficiency of the natural enemy is expected to be high in the monotypic habitat. Searching efficiency in the diverse stand depends on the level of "background noise" that heterospecific plants represent. If parasitoids can ignore non-relevant information easier in the MCS than in a

monoculture, e.g., if the parasitoid has a low responsiveness to heterospecific plants, they may be equally efficient in the MCS. If the perceived “background” noise is higher in the MCS, they are likely to be more successful in a monoculture.

Insect parasitoids can learn habitat cues and discriminate between signal and noise,^[29] a factor that can influence the parasitoids’ functioning in MCS, but which has not received much attention. Perfecto and Vet^[28] found that experience of foraging parasitoids with cues from mono- and dicultures led to differences in host-encounter rates. Therefore, it is likely that the individual responses of parasitoids to vegetation diversity depend on how the informational value of plant volatiles is perceived and used in a given multitrophic context and how that use is modified by experience.

Indirect Interactions in Simple and Complex Food Webs

Biological control has traditionally focused on maximizing the mortality of a target pest organism by one or a few natural enemy species, but how natural enemies and herbivores interact in the food web to influence mortality of a herbivore is little understood. Monocultures and MCS differ in the complexity of their food webs. The greater amount of resource subsidies, victims, pollen, nectar sources, and microclimates in MCS all contribute to a higher species richness of natural enemies in vegetationally diverse systems. Higher species richness of natural enemies increases the frequency of indirect interactions between natural enemies (Fig. 1). This may lead to a greater reduction of pest populations through facilitation between parasitoids and predators.^[30]

CONCLUSIONS

The effects of increased structural and infochemical complexity on natural enemy searching behavior and the possible facilitation of natural enemies in more complex food webs show that generalizations on how MCS affect herbivore–natural enemy interactions are difficult. The behavioral and biological details of the system determine whether MCS lead to persistence, pest outbreaks, or extinction of populations. To predict how given MCS influence parasitism rates in the field, a link between the behavioral and population level responses needs to be established.^[31] For example, some associated plants may increase populations of parasitoids by attracting and retaining individuals longer in the crop, whereas others may decrease populations in the target crops because of reduced immigration to or increased emigration from the crop.

Therefore, behavioral information on the responses of herbivores and their natural enemies to MCS may help us select specific plant species mixtures that suppress herbivores by both the direct negative bottom-up effects of vegetation diversity and the top-down effects of increased success of natural enemies.

REFERENCES

1. Power, M. Top-down and bottom-up forces in food-webs: do plants have primacy? *Ecology* **1992**, *73* (3), 733–746.
2. Ode, P.J.; Berenbaum, M.R.; Zangerl, A.R.; Hardy, C.W. Host plant, host plant chemistry and the polyembryonic parasitoid *Copidosoma sosares*: indirect effects in a tritrophic interaction. *Oikos* **2004**, *104* (2), 388–400.
3. Bukovinszky, T. *Tailoring Complexity; Multitrophic Interactions in Simple and Diversified Habitats*, Ph.D. Thesis. Wageningen University, Ponsen & Looijen BV: The Netherlands, 2004.
4. Smith, H.A.; McSorley, R. Intercropping and pest management: a review of major concepts. *Am. Entomol.* **2000**, *46* (3), 154–161.
5. Hooks, C.R.R.; Johnson, M.W. Impact of agricultural diversification on the insect community of cruciferous crops. *Crop Protect.* **2003**, *22* (2), 223–238.
6. Root, R.B. Organization of a plant–arthropod association in simple and diverse habitats: the fauna of collards (*Brassica oleracea*). *Ecol. Monogr.* **1973**, *43* (1), 95–124.
7. Sheehan, W. Response by specialist and generalist natural enemies to agroecosystem diversification: a selective review. *Environ. Entomol.* **1986**, *15* (3), 456–461.
8. Russell, E.P. Enemies hypothesis: a review of the effect of vegetational diversity on predatory insects and parasitoids. *Environ. Entomol.* **1989**, *18* (4), 590–599.
9. Price, P.W.; Bouton, C.E.; Gross, P.; McPheron, B.A.; Thompson, J.N.; Weis, A.E. Interactions among three trophic levels: influence of plants on interactions between insect herbivores and natural enemies. *Ann. Rev. Ecol. Syst.* **1980**, *11*, 41–65.
10. Vet, L.E.M.; Dicke, M. Ecology of infochemical use by natural enemies in a tritrophic context. *Ann. Rev. Entomol.* **1992**, *37*, 141–172.
11. Wilkinson, T.K.; Landis, D.A. Habitat diversification in biological control: the role of plant resources. In *Plant-Provided Food for Carnivorous Insects*; Wäckers, F.L., van Rijn, P.C.J., Bruin, J., Eds.; Cambridge University Press, *in press*.
12. Wäckers, F.L. The parasitoids’ need for sweets: sugars in mass rearing and biological control. In *Quality Control and Production of Biological Control Agents: Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI Publishing: Wallingford, 2003; 59–72.
13. Landis, D.A.; Wratten, S.D.; Gurr, G.M. Habitat management to conserve natural enemies of arthropod pests in agriculture. *Ann. Rev. Entomol.* **2000**, *45*, 175–201.
14. Kean, J.; Wratten, S.; Tylianakis, J.; Barlow, N. The population consequences of natural enemy enhancement,

- and implications for conservation biological control. *Ecol. Lett.* **2003**, *6* (7), 604–612.
15. Winkler, K.; Wäckers, F.L.; Valdivia, L.; Larraz, V.; Lenteren, J.C. Strategic use of nectar sources to boost biological control. *Landscape Management for Functional Biodiversity. Bull. IOBC WPRS* **2003**, *26* (4), 209–214.
 16. Dyer, L.E.; Landis, D.A. Influence of noncrop habitats on the distribution of *Eriborus tenebrans* (Hymenoptera: Ichneumonidae) in cornfields. *Environ. Entomol.* **1997**, *26* (4), 924–932.
 17. Need, J.T.; Burbutis, P.P. Searching efficiency of *Trichogramma nubilale*. *Environ. Entomol.* **1979**, *8* (2), 224–227.
 18. Gingras, D.; Dutilleul, P.; Boivin, G. Modeling the impact of plant structure on host-finding behaviour of parasitoids. *Oecologia* **2002**, *130* (3), 396–402.
 19. Huffaker, C.B. Experimental studies on predation: dispersion factors and predator–prey oscillations. *Hilgardia* **1958**, *27* (15), 343–383.
 20. Huffaker, C.B.; Shea, K.P.; Herman, S.G. Experimental studies on predation: complex dispersion and levels of food in an acarine predator–prey interaction. *Hilgardia* **1963**, *34* (9), 305–330.
 21. Kareiva, P. Habitat fragmentation and the stability of predator–prey interactions. *Nature* **1987**, *326*, 388–390.
 22. Kareiva, P.; Odell, G.M. Swarms of predators exhibit ‘preytaxis’ if individual predators use area-restricted search. *Am. Nat.* **1987**, *130* (2), 233–270.
 23. Ellner, S.P.; McCauley, E.; Kendall, B.E.; Briggs, C.J.; Hosseini, P.R.; Wood, S.N.; Janssen, A.; Sabelis, M.W.; Turchin, P.; Nisbet, R.M.; Murdoch, W.W. Habitat structure and population persistence in an experimental community. *Nature* **2001**, *412*, 538–543.
 24. Tumlinson, J.H.; Turlings, T.C.J.; Lewis, W.J. Semiochemically mediated foraging behavior in beneficial parasitic insects. *Arch. Insect Biochem. Physiol.* **1993**, *22* (3–4), 385–391.
 25. Monteith, L.G. Influence of plants other than the food plants of their host on host-finding by tachinid parasites. *Can. Entomol.* **1960**, *92* (9), 641–652.
 26. Dicke, M.; de Boer, J.G.; Höfte, M.; Rocha-Granados, M.C. Mixed blends of herbivore-induced plant volatiles and foraging success of carnivorous arthropods. *Oikos* **2003**, *101* (1), 38–48.
 27. Vet, L.E.M. Evolutionary aspects of plant–carnivore interactions. In *Insect–Plant Interactions and Induced Plant Defence*; Chadwick, D.J., Goode, J.A., Eds.; Wiley: Chichester, 1999; 3–13.
 28. Perfecto, I.; Vet, L.E.M. Effect of a nonhost plant on the location behavior of two parasitoids: the tritrophic system of *Cotesia* spp. (Hymenoptera: Braconidae), *Pieris rapae* (Lepidoptera: Pieridae), and *Brassica oleraceae*. *Environ. Entomol.* **2003**, *32* (1), 163–174.
 29. Vet, L.E.M.; Lewis, W.J.; Cardé, R.T. Parasitoid foraging and learning. In *Chemical Ecology of Insects*, 2nd Ed.; Bell, W., Cardé, R.T., Eds.; Chapman & Hall: London, 1995; 65–101.
 30. Cardinale, B.J.; Harvey, C.T.; Gross, K.; Ives, A.R. Biodiversity and biocontrol: emergent impacts of a multi-enemy assemblage on pest suppression and crop yield in an agroecosystem. *Ecol. Lett.* **2003**, *6* (9), 857–865.
 31. Vet, L.E.M. Parasitoid searching efficiency links behaviour to population processes. *Appl. Entomol. Zool.* **2001**, *36* (4), 399–408.

Natural Enemies and Biocontrol: Monitoring

Thomas W. Culliney

USDA, APHIS, PPQ, Center for Plant Health Science and Technology,
Raleigh, North Carolina, U.S.A.

INTRODUCTION

In pest management programs, natural enemy monitoring has two major functions: To follow the postrelease progress of introduced natural enemies in their successful establishment and subsequent dispersal and to evaluate the effectiveness of natural enemies in controlling their target pests. Assuming that careful screening of enemies has occurred, an additional function, to detect any negative impacts they might have on nontarget organisms, should not be necessary.

MONITORING POSTRELEASE ESTABLISHMENT OF NATURAL ENEMIES

In biological control programs, there are two basic ways in which natural enemies may be liberated.^[1] In an “open field release,” agents are released in the immediate vicinity of suitable hosts or prey, at which time their search behavior and orientation to hosts or prey are monitored. If enemies tend to disperse without attacking the target pest, they may be released into enclosures to confine them with the pest. Such a “confined release strategy,” using field cages, has often been used in the initial phase of natural enemy colonization. This method was used, with spectacular success, in the original colonization of the vedalia beetle, *Rodolia cardinalis* Mulsant, against the cottoncushion scale, *Icerya purchasi* Maskell, in California citrus groves in 1889. With confined release, even small numbers of biological control agents can be successfully established.

Recovery attempts are usually made soon after the initial release of natural enemies.^[1] Efforts are concentrated in time and space where the agents are most likely to be encountered. Failure to detect a second generation after release is generally an indication of poor physiological or ecological adaptation of the agent to its host or prey. However, even well-adapted species may be vulnerable to climatic extremes. For this reason, establishment of newly introduced natural enemies should be considered provisional until they have demonstrated their capacity to survive severe winter and summer conditions.

Ample evidence from successful biological control programs suggests that enemies that are destined to

exercise effective control over their hosts or prey will demonstrate this capacity soon after colonization. A fully effective agent should become easily and quickly established; conversely, failure of the natural enemy to establish in this manner indicates that it will never be a completely effective agent. An agent may be considered to be well established if it is found to be exerting measurable control within three host or prey generations to 3 years after its release.^[2] Certainly, releases may be reasonably discontinued after 3 years time if there is no evidence of the agent’s establishment.

EVALUATING THE EFFECTIVENESS OF NATURAL ENEMIES

Ideally, the evaluation of a biological control program commences before the introduction of natural enemies. A preintroduction pest population census establishes baseline data against which postintroduction pest densities are compared and the impact of natural enemies is assessed.^[3] Evaluation techniques are used mainly to ascertain whether biological control is occurring, whether it is sufficient to reduce pest populations to, and maintain them at, densities that are economically insignificant, and which natural enemies are involved. Other important reasons for evaluating natural enemies are to: 1) demonstrate the value and deficiencies of existing enemies, assess the need for introducing additional ones, and suggest the potential need to manipulate the environment or natural enemies to make the resident species more effective; 2) provide insights into the principles of population ecology; and 3) conclusively demonstrate the effectiveness of natural enemies in controlling pests to ensure continued support for biological control research.^[4] An additional reason that might be included here is to advance the theory and practice of biological control, which traditionally has been conducted in a trial-and-error manner, more as art than science.

Methods of Evaluation

Methods to assess the impact of natural enemies on pest populations fall into three major categories.^[1] An advantage of *qualitative methods* of evaluation is

that evidence gathered through frequent, detailed, and extensive field observations can provide the most rapid and least effort-intensive means of gaining some idea of the importance of natural enemies in the control of their hosts or prey. This observational method most often has been used to evaluate the effectiveness of newly introduced enemies (e.g., *R. cardinalis*, mentioned above). If establishment of the agent is followed by an obvious increase in its density, at the expense of its host or prey, followed by an obvious decrease in the latter's density, and this pattern is repeated as the enemy spreads into new areas, then there is strong presumptive evidence of cause (predation or parasitization) and effect (pest population decline).^[1] Pest-enemy population trends may lead a researcher to form an opinion of the effectiveness of the enemy. However, the researcher can never be completely certain that the observed average pest density was a result of natural enemy actions. There is always a possibility that the decrease in the pest's density after the establishment of the enemy was only a coincidence, that some other factor (e.g., a disease epizootic or adverse weather conditions) exerted its negative influence on the pest population in conjunction with establishment of the enemy, and that the enemy had little or nothing to do with the change in the status of the pest.

In using *quantitative methods*, attempts are made to show the effects of a natural enemy on a pest population by assessing various numerical measures. For example, percentage parasitization commonly has been used as a simple estimate of the efficacy of parasitoids. However, this measure cannot provide conclusive proof that the parasitoid is a regulative factor in its host's life cycle and thus is effecting control of its host. Both direct density dependence (functional response) and delayed density dependence (reproductive or numerical response) are essential elements of any assessment of the full regulative potential of a natural enemy.^[4] Two other common quantitative methods involve the correlation of density changes in natural enemy and pest populations and the analysis of life-table mortality data.

Correlation analysis

Periodic census of pest and natural enemy populations provides data on relative trends in pest and enemy densities, which can be graphed. Correlations between changes in these densities can then be analyzed, with the aim of assessing the effectiveness of the natural enemy in regulating the pest population (Fig. 1). Such data may be useful in indicating which environmental factors (independent variables), including the pest, are influencing the natural enemy population (dependent variable); however, they rarely, if ever, prove that the enemy is responsible for regulating the pest population at any particular average density.

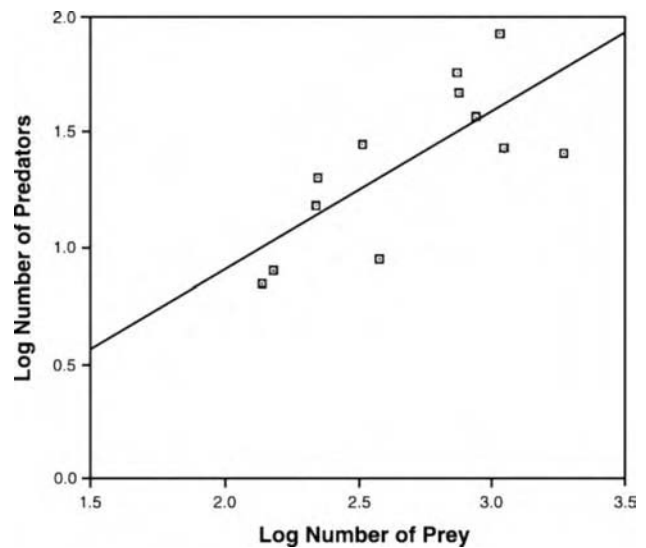


Fig. 1 Relationship between the densities of the predator *Leucopis* sp. (Diptera: Chamaemyiidae) and those of its prey, *Pineus pini* Macquart (Homoptera: Adelgidae), determined on each of the 12 survey dates. The significant, high degree of correlation ($r = 0.77$) indicates a density-dependent association between the two populations. (From Ref.^[5].)

Other factors, such as weather, edaphic conditions, or plant characteristics, may just as well be responsible for the observed pest-natural enemy trends. Nevertheless, the method continues to have utility in biological control studies, particularly in cases where the variables cannot be brought under the control of the investigator.^[5]

Life tables

Long used by actuaries for the computation of annuities and life insurance premiums, life tables were adapted to the study of animal populations, first by Deevey.^[6] A life table is a concise summary of certain vital statistics, such as mortality and survival, of a population. Traditionally, such tables were based on a cohort of 1000 individuals of the same generation. However, in the study of insect populations, data consist of variable field counts rather than a fixed, hypothetical number. For the simpler life tables usually constructed for insect populations, which comprise a set of periodic measurements of the population, in which changes from one census to the next are measured and, as far as possible, accounted for, the term "life budget" has been proposed.^[7] This term is probably a better descriptor; however, the original term continues in general use.

Basically, a typical life table consists of columns for age class (x , often expressed as stage for insect populations), survivorship (l_x , number of survivors entering the age interval), mortality (d_x , number dying within the age interval), and the mortality rate within the

age interval ($q_x = d_x/l_x$) (Table 1). A life table used to identify the relative contribution of various factors, including natural enemies, to the total mortality in an insect population would, in addition, include columns listing individual mortality factors (d_xF) and their numerical toll (d'_x), with a final column showing measures of “killing power” (k_x), calculated by subtracting $\log(l_x)$ from $\log(l_x + 1)$, the survivorship at

Table 1 A simple life table for a laboratory population of the aphid *Myzus persicae* Sulzer (Homoptera: Aphididae) on *Brassica oleracea*L. at 25°C

| <i>x</i> (day) | <i>l_x</i> | <i>d_x</i> | <i>q_x</i> |
|----------------|----------------------|----------------------|----------------------|
| 1 | 27 | 0 | 0 |
| 2 | 27 | 0 | 0 |
| 3 | 27 | 0 | 0 |
| 4 | 27 | 0 | 0 |
| 5 | 27 | 0 | 0 |
| 6 | 27 | 0 | 0 |
| 7 | 27 | 0 | 0 |
| 8 | 27 | 0 | 0 |
| 9 | 27 | 1 | 0.037 |
| 10 | 26 | 1 | 0.038 |
| 11 | 25 | 0 | 0 |
| 12 | 25 | 0 | 0 |
| 13 | 25 | 0 | 0 |
| 14 | 25 | 3 | 0.120 |
| 15 | 22 | 2 | 0.091 |
| 16 | 20 | 2 | 0.100 |
| 17 | 18 | 0 | 0 |
| 18 | 18 | 1 | 0.056 |
| 19 | 17 | 4 | 0.235 |
| 20 | 13 | 3 | 0.231 |
| 21 | 10 | 0 | 0 |
| 22 | 10 | 2 | 0.200 |
| 23 | 8 | 0 | 0 |
| 24 | 8 | 0 | 0 |
| 25 | 8 | 1 | 0.125 |
| 26 | 7 | 2 | 0.286 |
| 27 | 5 | 1 | 0.200 |
| 28 | 4 | 0 | 0 |
| 29 | 4 | 0 | 0 |
| 30 | 4 | 0 | 0 |
| 31 | 4 | 1 | 0.250 |
| 32 | 3 | 0 | 0 |
| 33 | 3 | 1 | 0.333 |
| 34 | 2 | 1 | 0.500 |
| 35 | 1 | 1 | 1.000 |

(From Ref.^[13].)

the beginning of the succeeding age interval. These k values are analyzed to gauge the relative importance of the mortality factors and to identify key factors contributing to the observed changes in density in the population.^[8] In gathering data for a life table, samples are taken of the age classes, egg to adult, from the entire pest population over the course of a season. Sampling should be conducted over several pest generations and in various locations.

Varley and Gradwell^[9] devised the technique known as key factor analysis, which uses a graphical approach to analyze the environmental factors having the greatest influence on intergenerational population dynamics. All k values are summed over each generation to give a total generational mortality, K . Total K , along with each individual k value, is plotted over several generations. The relative importance of each k graph is readily apparent from its similarity to the graph of total K . The mortality factor that most closely correlates to total generational mortality constitutes the key mortality factor. The degree of density dependence operative in the mortality factors impinging on a population can be revealed by plotting each k value against the corresponding survivorship.^[8] However, detection of density dependence in the mortality caused by a natural enemy is not sufficient to admit the conclusion that the enemy’s actions are regulating. Such action can only be conclusively demonstrated by using experimental techniques.

Experimental methods, which involve active manipulation of the interacting populations, can provide the necessary proof of pest population regulation by natural enemies.^[4] Three principal methods have been employed.^[1,3,4,10,11]

Addition method

This method seeks to measure the impact of new natural enemies. It involves “before-and-after” comparisons of similar plots, some receiving natural enemies, others not. Differences in pest densities between plots receiving natural enemies and those not receiving natural enemies can be attributed to action by the enemies. Same-plot photographs taken before and after the introduction of the natural enemy are commonly used in this method, and may provide better evidence of success than population census data. In particular, in some biological weed control programs, success has been spectacularly demonstrated by before-and-after photographs (e.g., in the control of prickly pear, *Opuntia stricta* Haworth, in Australia, and St. Johnswort or Klamath weed, *Hypericum perforatum* L., in California^[4]).

Exclusion method

The exclusion, or subtraction, method was pioneered by Smith and DeBach.^[12] The method is employed

after natural enemies are well established, and involves exclusion or elimination of natural enemies from plots or other experimental units (e.g., individual plants or parts thereof, such as branches or leaves), which are then compared with their counterparts, to which enemies retain access. Resulting differences in pest equilibrium densities between the two treatments reveal the regulatory effectiveness of the enemy. Exclusion of natural enemies may be accomplished by mechanical means, including cages (most frequently used), moats, other barriers, or hand removal, or by the use of pesticides that selectively kill the enemies without harming the pest (“insecticidal check technique”). Experiments must be designed to ensure that only the variable of interest, i.e., the presence of the natural enemy, is manipulated, and that the exclusion technique itself is not exerting any appreciable influence on the populations.

Interference method

The interference, or neutralization, method involves reducing the efficiency of natural enemies in one set of plots while leaving them undisturbed in another. Various means have been used, such as the insecticidal check, biological check (ant interference with natural enemies attacking honeydew-producing insects), hand removal, and trap techniques (pesticidal treatment of areas surrounding test plots to kill dispersing enemies). All either kill, exclude, or otherwise disturb a large proportion of the enemies originally present, resulting in a reduction in the average rate of increase of the enemy density with respect to that of the pest. Removed from predation pressure, the pest population can attain a higher average density, which demonstrates that natural enemies were responsible for regulating it at the original, lower density. The theoretical basis for the method derives from the pest resurgences that commonly follow pesticidal applications to crops, which can decimate natural enemy populations.

CONCLUSION

Probably the most important reason for monitoring natural enemies is to evaluate their efficacy as biological control agents. As Luck^[3] pointed out, biological control is the foundation of pest management, and thus its evaluation is an important endeavor, one that has all too frequently been ignored. Traditionally, a biological control program, which might have substantial startup costs and take years to implement, was considered a success if the pest problem it was designed to combat ceased to be of any further economic importance (a conclusion based obviously on qualitative evidence); because effective control of the pest was “obvious,” continued monitoring of natural enemies

was not deemed necessary, and attention shifted to the next pest problem. Alternatively, if the problem persisted because natural enemies failed to establish or otherwise were ineffective in controlling the pest, further efforts to determine reasons for the failure usually were not considered a wise application of scarce resources—money and manpower could be put to better use elsewhere. This lack of scientific rigor in the conduct of biological control has long been deplored, particularly by academic researchers, who have striven to put the discipline on a more sound scientific footing.

All of the methods developed to evaluate natural enemies have weaknesses. The observational data gathered using qualitative techniques completely lack statistical power, and any conclusions drawn from them constitute sheer opinion. With the quantitative methods, it is critical that thorough, representative sampling of populations be performed, lest erroneous conclusions be drawn. However, even with adequate sampling, these methods provide, at best, mere indications of the control potential of enemies. Although correlation analysis may indicate a highly significant relationship between two interacting variables, it cannot conclusively establish cause and effect. In particular, the variety of environmental influences affecting populations in the field render the use of correlative techniques unreliable in evaluating the true role of natural enemies in regulating pest populations.

Life tables provide a quantitative framework within which to examine the sources of mortality, and their magnitude, acting on a population. Analysis of life tables seeks to determine the relative importance of the mortality factors as they influence a population’s dynamics and to predict future population trends. Again, the validity of the conclusions rests on the assumption that an adequate, representative sample has been taken. Whereas life tables can provide valuable demographic information on natural enemies, their hosts or prey, and other environmental factors, sampling a faunal complex in various habitats and determining accurately the severity of mortality caused by various factors often require so much work as to make the method impractical, even if it should indicate the effectiveness of a particular mortality factor in population regulation. In any case, absolute proof that any particular factor is responsible for maintaining a pest population at a lower density than would be the case were the factor absent would be difficult to obtain.

Of the available methodology, the experimental techniques, designed to eliminate extraneous influences as far as possible, offer the most scientifically robust means of evaluating the control potential of natural enemies. Exclusion methods have been considered to be the most useful by researchers, although they in practice are not without their drawbacks. As with all experimental

outcomes, results of exclusion experiments should be interpreted with caution. However, even with their potential to bias results, exclusion techniques remain excellent (and proven) means to assess the effectiveness of natural enemies in controlling pest populations.

REFERENCES

1. DeBach, P.; Bartlett, B.R. Methods of colonization, recovery and evaluation. In *Biological Control of Insect Pests and Weeds*; DeBach, P., Ed.; Chapman and Hall Ltd.: London, 1964; 402–426.
2. Clausen, C.P. The time factor in biological control. *J. Econ. Entomol.* **1951**, *44* (1), 1–9.
3. Luck, R.F. Techniques for studying the impact of natural enemies. In *Biological Control: Issues in the Tropics*; Ooi, P.A.C., Lim, G.S., Teng, P.S., Eds.; Malaysian Plant Protection Soc.: Kuala Lumpur, 1992; 69–83.
4. DeBach, P.; Huffaker, C.B.; MacPhee, A.W. Evaluation of the impact of natural enemies. In *Theory and Practice of Biological Control*; Huffaker, C.B., Messenger, P.S., Eds.; Academic Press: New York, 1976; 255–285.
5. Culliney, T.W.; Beardsley, J.W., Jr.; Drea, J.J. Population regulation of the Eurasian pine adelgid (Homoptera: Adelgidae) in Hawaii. *J. Econ. Entomol.* **1988**, *81* (1), 142–147.
6. Deevey, E.S., Jr. Life tables for natural populations of animals. *Quart. Rev. Biol.* **1947**, *22* (4), 283–314.
7. Richards, O.W. The theoretical and practical study of natural insect populations. *Annu. Rev. Entomol.* **1961**, *6*, 147–162.
8. Horn, D.J. *Ecological Approach to Pest Management*; Guilford Press: New York, 1988.
9. Varley, G.C.; Gradwell, G.R. Key factors in population studies. *J. Anim. Ecol.* **1960**, *29* (2), 399–401.
10. Luck, R.F.; Shepard, B.M.; Kenmore, P.E. Experimental methods for evaluating arthropod natural enemies. *Annu. Rev. Entomol.* **1988**, *33*, 367–391.
11. DeBach, P.; Huffaker, C.B. Experimental techniques for evaluation of the effectiveness of natural enemies. In *Biological Control*; Huffaker, C.B., Ed.; Plenum Press: New York, 1971; 113–140.
12. Smith, H.S.; DeBach, P. The measurement of the effect of entomophagous insects on population densities of their hosts. *J. Econ. Entomol.* **1942**, *35* (6), 845–849.
13. Culliney, T.W.; Pimentel, D. The intrinsic rate of natural increase of the green peach aphid, *Myzus persicae* (Sulzer) (Homoptera: Aphididae), on collards (*Brassica oleracea* L.). *Can. Ent.* **1985**, *117* (9), 1147–1149.

Natural Enemies and Biocontrol: Quality Control Guidelines and Testing Methods

Norman C. Leppla

*Department of Entomology and Nematology, Institute of Food and Agricultural Sciences,
University of Florida, Gainesville, Florida, U.S.A.*

Barbra C. Larson

*Department of Environmental Sciences, Institute of Food and Agricultural Sciences,
University of Florida, Gainesville, Florida, U.S.A.*

INTRODUCTION

Commercial producers and government organizations have developed quantitative quality control methods for large-scale production of natural enemies.^[1,2] These methods have evolved to assure the reliable production of natural enemies that meet the performance standards and expectations of customers and government clientele groups. Facilities, equipment, materials, and standard operating procedures are monitored for consistency, and the quality of natural enemies is evaluated during and after production. This evaluation is accomplished by means of standardized criteria and tests for each species, typically including rate of development, survival, identity, size, weight, and essential behavior. Natural enemies must disperse, search the habitat, and successfully consume or parasitize hosts. Quality control methods enable producers and users to predict and confirm the performance of natural enemies.

IMPETUS FOR DEVELOPMENT OF QUALITY CONTROL GUIDELINES

In addition to the marketplace and government clientele, regulatory authorities are continuing to require some proof of natural enemy identity, purity, and efficacy. Quality control methods are used by producers of natural enemies to identify the species in each shipment, guarantee that no contaminants are present, and provide evidence that the target pest can be controlled without unacceptable side effects. This is analogous to the labeling requirements for chemical and biological pesticides. However, unlike many other countries, the United States does not require efficacy data for multicellular natural enemies, i.e., arthropods and nematodes, but individual states can impose more stringent regulations.

The threat of increasingly restrictive and expensive regulation has caused producers and suppliers of

commercial natural enemies to form trade organizations. The Association of Natural Bio-control Producers (ANBP) represents about 40 producers and distributors located across the United States and several European and Canadian companies that have significant markets in North America (<http://www.anbp.org>). The Association of Natural Bio-control Producers was founded in 1990 to foster collaboration among the member companies and help the industry prosper. Particular attention has been placed on quality issues and research and education in the development and use of biological control products. The Association of Natural Bio-control Producers standards require that the product label include the species, number of individuals in the package, packing date, level of purity, and number of living natural enemies. The European counterpart to ANBP is the International Biocontrol Manufacturers Association (IBMA), founded in 1995 to address microbial and macrobial natural enemies, pheromones, and natural products (<http://www.ibma.org>). Together, they have encouraged the regulatory community and the Organization for Economic Cooperation and Development to facilitate commercial biological control. The leaders of ANBP and IBMA collaborate with each other and the International Organization for Biological Control (IOBC), Arthropod Mass Rearing and Quality Control Working Group (AMRQC).^[3] These organizations have developed quality control guidelines for more than 40 natural enemies (Table 1).

TESTS AND METHODS

Colleagues from ANBP, IBMA, and AMRQC, with considerable support from the European Union (EU) and United States Department of Agriculture (USDA, Agricultural Research Service, Animal and Plant Health Inspection Service, and Cooperative State Research, Education and Extension Service), assembled the international quality control standards or guidelines

Table 1 List of natural enemies that have some quality control standards

| Natural enemy | Family: Order |
|--|--------------------------------|
| <i>Amblyseius (Neoseiulus) degenerans</i> Berlese | Acarina: Phytoseiidae |
| <i>Anthocoris nemoralis</i> (Fabricius) | Hemiptera: Anthocoridae |
| <i>Aphelinus abdominalis</i> Dalman | Hymenoptera: Aphelinidae |
| <i>Aphidius colemani</i> Viereck | Hymenoptera: Braconidae |
| <i>Aphidius ervi</i> (Haliday) | Hymenoptera: Braconidae |
| <i>Aphidoletes aphidimyza</i> (Rondani) | Diptera: Cecidomyiidae |
| <i>Aphytis lingnanensis</i> Compere | Hymenoptera: Aphelinidae |
| <i>Aphytis melinus</i> DeBach | Hymenoptera: Aphelinidae |
| <i>Chrysoperla carnea</i> Steph. | Neuroptera: Chrysopidae |
| <i>Chrysoperla rufilabris</i> (Burmeister) | Neuroptera: Chrysopidae |
| <i>Cryptolaemus montrouzieri</i> Mulsant | Coleoptera: Coccinellidae |
| <i>Dacnusa sibirica</i> Telenga | Hymenoptera: Braconidae |
| <i>Dicyphus hesperus</i> Wagner | Hemiptera: Miridae |
| <i>Diglyphus isaea</i> (Walker) | Hymenoptera: Eulophidae |
| <i>Encarsia formosa</i> Gahan | Hymenoptera: Aphelinidae |
| <i>Eretmocerus eremicus</i> (Rose) | Hymenoptera: Aphelinidae |
| <i>Eretmocerus mundus</i> Mercet | Hymenoptera: Aphelinidae |
| <i>Galendromus occidentalis</i> (Nesbitt) | Acarina: Phytoseiidae |
| <i>Goniozus legneri</i> Gordh | Hymenoptera: Bethyidae |
| <i>Hypoaspis miles</i> Berlese | Acarina: Laelapidae |
| <i>Leptomastix dactylopii</i> Howard | Hymenoptera: Encyrtidae |
| <i>Macrolophus caliginosus</i> Wagner | Hemiptera: Miridae |
| <i>Mesoseiulus longipes</i> (Evans) | Acarina: Phytoseiidae |
| <i>Muscidifurax raptor</i> Girault and Sanders | Hymenoptera: Pteromalidae |
| <i>Muscidifurax zaraptor</i> Kogan and Legner | Hymenoptera: Pteromalidae |
| <i>Neoseiulus californicus</i> McGregor | Acarina: Phytoseiidae |
| <i>Neoseiulus cucumeris</i> (Oudemans) | Acarina: Phytoseiidae |
| <i>Orius</i> spp. (<i>O. aldibipennis</i> , <i>O. insidiosus</i> , <i>O. laevigatus</i> , <i>O. majusculus</i>) | Hemiptera: Anthocoridae |
| <i>Pentalitomastix plethorica</i> Caltagirone | Hymenoptera: Encyrtidae |
| <i>Phytoseiulus persimilis</i> Athias-Henriot | Acarina: Phytoseiidae |
| <i>Podisus maculiventris</i> Say | Hemiptera: Pentatomidae |
| <i>Spalangia nigroaenea</i> Curtis | Hymenoptera: Pteromalidae |
| <i>Trichogrammatoidea bactrae</i> Nagaraja | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma brassicae</i> Bezd. (= <i>T. maidis</i>) | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma cacoeciae</i> Marchal | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma dendrolimi</i> Matsumura | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma minutum</i> Riley | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma platneri</i> Nagarkatti | Hymenoptera: Trichogrammatidae |
| <i>Trichogramma pretiosum</i> Riley | Hymenoptera: Trichogrammatidae |
| <i>Thripobius semiluteus</i> Boucek | Hymenoptera: Eulophidae |
| <i>Steinernema carpocapsae</i> (Weiser) | Rhabditida: Steinernematidae |

Incorporates information from Ref.^[5].

for natural enemies from the ANBP labels and Product Profiles^[4] and the IOBC/EU guidelines. Recently, the American Society for Testing and Materials approved guidelines for selected species. The guidelines include quality control standards and tests based on specified criteria, e.g., quantity, sex ratio, emergence, fecundity, longevity, parasitism, predation, size, and performance in the laboratory and field. The tests are constantly updated (<http://www.amrqc.org>) and used in both commercial and government production facilities. Proprietary quality control tests used in independent organizations are probably similar to those in the public domain.

The well-established quality control testing protocol for *Trichogramma brassicae* Bezd. serves as a typical example. Test conditions include $23 \pm 2^\circ\text{C}$, $75 \pm 10\%$ RH, and a 16-hour light/8-hour dark photoperiod. Molecular techniques are used to verify the species once each year, requiring 30 fresh specimens. For sex ratio tests, with a standard of $\geq 50\%$ females, 100 adults are assessed from a specified number of release units. Each female is expected to produce ≥ 40 offspring every 7 days, and 80% of the females should live for at least 7 days ($n = 30$ per month or batch). The required rate of parasitism is ≥ 10 hosts per female

every 4 hr. Methods are specified for holding 24-hr-old females and counting the number of embryonated eggs, both factitious [*Ephesia kuehniella* Zeller and *Sitotroga cerealella* (Oliver)] and natural [*Ostrinia nubilalis* (Hübner)]. Egg masses are used, and host-cluster acceptance should be $\geq 80\%$ because, often, a parasitoid finds only one egg mass during its lifetime.^[5]

FUTURE APPLICATIONS

The quality of natural enemies is improving as associated research and technology advances. Molecular techniques can be used to rapidly identify species and strains, screen colonies for pathogens, and, eventually, genetically engineer natural enemies. Artificial diets have been developed by publicly funded researchers, patented and licensed for use in rearing predatory insects. These diets have been incorporated into freeze-dried food packets that extend the shelf life of patented predator products. Automated encapsulation of diets may enable the efficient mass production of egg predators and parasitoids. The American Society for Testing and Materials has accepted natural enemies into its product quality program, and the International Standards Organization has modified its structure to accommodate insect rearing and similar technologies. The Arthropod Mass Rearing and Quality Control Working Group has reestablished the connections between natural enemy rearing and the resulting quality because they are interdependent. These advancements and others in the near future will continue to require investments in quality control research and implementation.

CONCLUSION

Quality control of natural enemies was featured at a recent IOBC conference.^[6] Resolutions were adopted to develop new technologies for augmentation biological control, including the mass production, formulation, and delivery of high-quality natural enemies. Regulatory authorities were encouraged to develop science-based laws and procedures, if they plan to register biological control products, and to take into account the relative importance and long history of safe use of biological control. A final resolution encouraged governments to support biological control research and development that will assure a supply

of high-quality natural enemies. The continued development of quality control methods and performance standards by commercial producers and government organizations will provide greater consistency in the application of augmentation biological control.

ACKNOWLEDGMENTS

The following members of ANBP, IBMA, and IOBC have made significant contributions to the first author's understanding of industry issues over the years: Carol Glenister (IPM Laboratories), Karel Bolckmans (Koppert Biological Systems), Ernest Delfosse (USDA, Agricultural Research Service), Joop van Lenteren (Laboratory of Entomology, Wageningen University), Sinthya Penn (Beneficial Insectary), Angela Hale (The Bug Factory), Dan Cahn (Syngenta Bio-line, Inc.), and Don Elliott (Applied Bio-nomics, Ltd.).

REFERENCES

1. van Lenteren, J.C., Ed.; *Quality Control and Production of Biological Control Agents, Theory and Testing Procedures*; CABI Publishing: Cambridge, MA, 2003.
2. Leppla, N.C. The foundation of quality control for insect rearing. In *Principles and Procedures for Insect Rearing*; Schneider, J., Ed.; Mississippi State University Press: Mississippi State, MS, *in press*.
3. Leppla, N.C., Bloem, K.A., Luck, R.F., Eds.; *Quality Control for Mass-Reared Arthropods, Proceedings of the Eighth and Ninth Workshops of the International Organization for Biological Control Working Group on Quality Control of Mass-Reared Arthropods*; University of Florida, Institute of Food and Agricultural Sciences: Gainesville, FL, 2002.
4. Penn, S.L.; Ridgway, R.L.; Scriven, G.T.; Inscoc, M.N. Quality assurance by the commercial producer of arthropod natural enemies. In *Mass-Reared Natural Enemies: Application, Regulation, and Needs*; Ridgway, R.L., Hoffman, M.P., Inscoc, M.N., Glenister, C.S., Eds.; Proc. Thomas Say Publ. in Entomol.; Entomol. Soc. Amer.: Lanham, MD, 1998; 202–230.
5. van Lenteren, J.C.; Hale, A.; Klapwijk, J.N.; van Schelt, J.; Steinberg, S. Guidelines for quality control of commercially produced natural enemies. In *Quality Control and Production of Biological Control Agents, Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI Publishing: Cambridge, MA, 2003; 265–303.
6. Aeschlimann, J.P. Proceedings of the IOBC montpellier conference. *Entomophaga* **1996**, *41*, 307–531.

Natural Enemies: Destruction by Pesticides

Joseph D. Cornell

College of Environmental Science and Forestry, State University of New York,
Syracuse, New York, U.S.A.

INTRODUCTION

Natural enemies are the coevolved predators, parasites, and even competitors that tend to keep populations of all organisms in check. In the absence of these natural enemies some species can become serious pests. Unfortunately, pesticides intended to control pest populations often also have the undesired effect of harming populations of natural enemies. When this happens, pest species can actually rebound beyond the ability of chemical pesticides alone to control. Pesticides can harm populations of natural enemies in many ways including bioaccumulation in predators and biomagnification from one trophic level to the next, potentially leading to cascade effects in the affected ecosystem. These effects and more are described with examples to show how a consideration of populations of natural enemies should be an essential part of all pest control efforts, particularly if those efforts include traditional chemical pesticides.

DESTRUCTION OF NATURAL ENEMIES BY PESTICIDES

Pesticides are designed to kill or disrupt the life cycle of pest species. Too often, however, they also have the unintended effect of killing or harming useful species which are the natural enemies of pests. Natural enemies are the predators, parasites, and even competitors, which act as a biological control on pest populations. Natural enemies include vertebrate species, invertebrate species, and even microorganisms such as fungi and bacteria. In the absence of natural enemies, pest populations can explode. Currently, the greatest threat to populations of natural enemies is from traditional, chemical pesticides.

Because many chemical pesticides are lethal or toxic to a broad spectrum of organisms, pesticides can harm both pests and non-pest species, including natural enemies. Indeed, pesticides can often have greater effects on populations of natural enemies than on pest populations. Predators that feed on pest species may receive higher doses of pesticides due to **bioaccumulation**,^[1] and, due to their smaller populations relative to populations of their prey, predator populations

can take much longer to rebound, allowing pest species to grow unchecked. Harm to predator populations can then actually lead to a **resurgence** of pest populations following the application of pesticides.^[2] For example in England, attempts to control the cabbage aphid (*Brevicoryne brassicae*) with commercially applied pesticides in 1956 caused more harm to the aphid's natural enemies than to the aphid and quickly resulted in the largest outbreak of cabbage aphid ever seen.^[3]

In addition to eliminating predators or parasites that help to control pest populations, the use of pesticides can eliminate competitors and permit the emergence of **secondary pest** populations. In the 1950s in Central America there were two major insect pests of cotton, which were being controlled successfully with fewer than five applications of organochloride and organophosphate pesticides per year.^[4] By 1955, three new secondary cotton pests had emerged in response to reduced competition from the first two pests. To control all five pests, applications of pesticides increased to 10 times per year. By the 1960s there were a total of eight major pests of cotton and pesticide applications had increased on average to 28 times per year.

Perhaps the most important example of the emergence of a secondary pest following the loss of natural enemies is that of the brown planthopper, (*Nilaparvata lugens*). Until the 1970s the brown planthopper was a relatively unimportant pest of rice throughout Southeast Asia and the Pacific.^[5] The widespread application of modern pesticides eliminated not only the primary pests of rice, but also the spiders and predatory insects that fed upon these pests.^[6] Because the brown planthopper lays its eggs between densely packed stems of rice, however, this particular pest was more difficult to control with chemical pesticide sprays.^[7] As a result of the loss of its competitors and other natural enemies, populations of the brown planthopper grew dramatically in India, China, Indonesia, and throughout the rest of Southeast Asia and the Pacific during the 1970s and the 1980s.

(To see what the brown planthopper and its eggs look like, as well as an image of what "hopperburn" caused by this pest looks like, go to <http://www.iclarm.org/irri/Troprice/html/I-bphopper.htm>.)

The brown planthopper is a classic example of an r-selected pest species.^[8] Instead of competing with

other species, the brown planthopper avoids competitors and evades predators by reproducing rapidly and through long-range dispersal. Each female lays between 200 and 300 eggs within a 2-week period producing a new generation every 3 to 4 weeks. Every second generation produces winged individuals, which allows them to disperse over great distances. Under natural conditions, these traits do not protect individuals from predation, but allow the species to survive by rapidly moving ahead of predators from one field to the next.

In the absence of predators and competition from other pest species, the brown planthopper emerged as the most important rice pest in Asia. In response to this new threat to rice production, the International Rice Research Institute (IRRI) in the Philippines developed new breeds of rice that were resistant to the brown planthopper.^[7] These new breeds of rice incorporated genes that gave the rice plants some resistance to the brown planthopper. However, due to rapid rates of reproduction, the brown planthopper was able to adapt quickly to these new breeds of rice leading to the **resurgence** of brown planthopper populations. In this case, resurgence occurs when a pest species adapts to pesticides or to pest-resistant crops. Unless the pest is entirely eliminated, the net effect is to create a new population of pests with genes for improved resistance to the pesticide or crop.

It eventually became clear that new pesticides and pest-resistant crops could not be developed rapidly enough to control the brown planthopper. In Indonesia, one of the nations hardest hit by the brown planthopper, it was decided to limit the use of pesticides so that populations of natural enemies would rebound. Accordingly, in 1986 Indonesia severely restricted the use of 57 different pesticides—primarily organochloride and organophosphate pesticides—on rice. Only carbamates, which are used as herbicides primarily,^[9] and juvenile hormone insecticides were permitted for general application on rice. The use of these restricted pesticides on rice was limited to areas where infestations of insect pests exceeded specific thresholds.

In addition to reducing its reliance on pesticides, Indonesia committed itself to implementing methods based on Integrated Pest Management (IPM), which emphasizes biological and cultural practices to control pests. With the help of the FAO, Indonesia trained 2.5 million farmers in the application of IPM methods,

and in 1988 Indonesia eliminated all economic subsidies for pesticides. These new policies have been remarkably effective and crop losses due to the brown planthopper have decreased. It is widely accepted now that the emergence of the brown planthopper as the major pest of rice in Asia was due to the use of pesticides, which harmed populations of the brown planthopper's natural enemies. In the case of the brown planthopper therefore, ironically, controlling the use of pesticides was the key to controlling the pest.

For more on the pesticides used to control the brown planthopper on Rice, go to <http://www.iclarm.org/irri/Troprice/>, and follow the links to "Insect Management," "Pests and when they are important," and then "Brown Planthopper."

REFERENCES

1. Carpenter, S.R. Ecosystem ecology: integrated physical, chemical and biological processes. In *Ecology*; Dodson, S.I., Allen, T.F.H., Carpenter, S.R., Ives, A.R., Jeanne, R.L., Kitchell, J.F., Langston, N.E., Turner, M.G., Eds.; Oxford University Press: New York, 1998; 434 pp, Chapter 14.
2. Metcalf, R.L. The ecology of insecticides and the chemical control of insects. In *Ecological Theory and Integrated Pest Management Practice*; Kogan, M., Ed.; John Wiley and Sons: New York, 1986; 251–297.
3. Ripper, W.E. Effects of pesticides on balance of arthropod populations. *Annu. Rev. Entomol.* **1956**, *1*, 403–438.
4. Flint, M.L.; van den Bosch, R. *Introduction to Integrated Pest Management*; Plenum Press: New York, 1981.
5. IRRI. *Brown Planthopper: Threat to Rice Production in Asia*; International Rice Research Institute: Los Baños, Philippines, 1979.
6. IRRI. *Friends of the Rice Farmer: Helpful Insects, Spiders, and Pathogens*; International Rice Research Institute: Los Baños, Philippines, 1987.
7. Fox, J.J. Managing the ecology of rice production in Indonesia. In *Indonesia: Resources, Ecology, and Environment*; Hardjono, J., Ed.; Oxford University Press: Oxford, 1991; Chapter 4.
8. Conway, G. Man versus pests. In *Theoretical Ecology*; May, R.M., Ed.; Blackwell Scientific Publications: Oxford, 1981; 356–386.
9. Watson, J.E. Pesticides as a source of pollution. In *Pollution Science*; Pepper, I.L., Gerba, C.P., Brusseau, M.L., Eds.; Academic Press: New York, 1996; Chapter 17.

Natural Vegetation Management to Improve Parasitoids in Farming Systems

Giovanni Burgio
Alberto Lanzoni
Antonio Masetti

Dipartimento di Scienze e Tecnologie Agroambientali-Entomologia, Alma Mater Studiorum
Università di Bologna, Bologna, Italy

INTRODUCTION

Conservation biological control involves environmental manipulation to enhance the fecundity and longevity of natural enemies, modify their behaviour, and provide shelter from adverse environmental conditions. Many authors have pointed out the potential importance of vegetation management to conserve and augment natural enemies of arthropod pests, including parasitoids, in agriculture. Recently, in many countries, the promotion of floristic diversity within farming systems has become an aim of agricultural policy.

In this contribution, attention will be focused on the role of natural vegetation in enhancement biological control (conservation biological control *sensu strictu*) by parasitoids against pest arthropods in agriculture.

ECOLOGICAL FUNCTIONS OF NATURAL VEGETATION ON PARASITIDS

Natural vegetation, including weeds, shrubs, and hedgerows, offers different requisites for parasitoids: adult food sources, alternative hosts, and physical refuge.^[1] Natural vegetation can also provide microhabitats that usually are unavailable in monocultures, providing shelter for overwintering insects and ecological corridors for beneficials, including insect parasitoids.^[2-6]

It is demonstrated that the survival and activity of parasitoids are strictly influenced by the availability and quality of food (nectar, pollen, and honeydew). Some experiments pointed out that immature females of parasitoids are attracted by flowers to mature their eggs.^[3]

The literature contains many reviews on the relationships between vegetation management and parasitoid efficiency (Table 1). Many cases of increased parasitism rate due to the presence of adult food sources and/or alternative hosts provided by natural vegetation have been listed and reviewed.^[1-6]

It is reported that the mean longevity of *Diadegma semiclausum*, a parasitoid of *Plutella xylostella*, was

significantly greater when feeding on buckwheat flowers as opposed to water only;^[1] buckwheat and the diluted honey treatment did not differ statistically, making buckwheat a potential species for enhancing longevity. The results of previous research also suggest that buckwheat could be a potential plant for enhancing the fecundity of parasitoid species. It is demonstrated that flowers can increase longevity also in *Microtonus hyperodae*, a parasitoid of pasture pest.^[1]

A classical example of increased natural enemy effectiveness due to the presence of alternative hosts is the *Anagrus* sp.–grapevine leafhopper system, studied in California and Europe.^[1-6] The western grape leafhopper *Erythroneura elegantula* is a major pest of grapes in many regions of the western United States. *Anagrus epos* is an important and effective egg parasitoid of *E. elegantula*, which overwinters inside leafhopper eggs. *E. elegantula* passes the winter in the adult stage; thus the parasitoid needs alternate hosts for successful overwintering. If alternate hosts for *A. epos* near the vineyard are lacking, the parasitoid must migrate, releasing *E. elegantula* from an important mortality factor in early spring and allowing the leafhopper to reach pest status. Wild plants such as blackberry (*Rubus* spp.), as well as cultivated French prune (*Prunus domestica*), support eggs of alternative leafhopper host. Vineyards situated downwind of these plants have higher early-season *A. epos* parasitism that contributes to grape leafhopper control. A similar situation has been observed in Switzerland and Italy, where the green grape leafhopper *Empoasca vitis*, which overwinters in the adult stage as well, suffers from a higher level of egg parasitism in vineyards near bramble (*Rubus ulmifolius*) or holly oak (*Quercus ilex*), harboring overwintering eggs of alternative leafhopper hosts of the parasitoid *Anagrus atomus*.

Perennial plants such as trees and shrubs can offer alternative hosts for other trophic systems. For example, in northern Italy rural landscape, seven species of braconid parasitoids were sampled on parasitized aphids infesting blackthorn (*Prunus spinosa*) within hedgerows; to a lesser extent, also willow

Table 1 Summary of applied studies demonstrating increasing parasitization due to natural vegetation and noncrop plant

| Cropping system | Natural vegetation (including landscape structure) | Pest | Parasitoids |
|-------------------|--|---|---|
| Alfalfa | Wild flowers | <i>Colias philodice</i> | <i>Apanteles medicaginis</i> (Braconidae) |
| Apple | <i>Phacelia</i> spp. <i>Eryngium</i> spp. | Aphids | <i>Aphelinus mali</i> (Aphelinidae) |
| Apple | Wild flowers | Tent caterpillar <i>Malacosoma americanum</i> Codling moth <i>Cydia pomonella</i> | Various species of parasitic wasps |
| Apple | Weeds | Apple maggot <i>Rhagoletis pomonella</i> | Braconidae |
| Cabbage | <i>Crataegus</i> sp. | Diamondback moth <i>Plutella maculipennis</i> | <i>Horgenes</i> sp. (Campopleginae) |
| Cereals | Wild flowers | Aphids | Aphidiinae (Braconidae) |
| Cereals | Nearby natural habitats | <i>Eurygaster integriceps</i> | Scelionidae |
| Cotton | Flax <i>Linum</i> spp. | <i>Heliothis virescens</i> | <i>Archytas</i> spp. (Tachinidae) |
| Cotton | Ragweed <i>Ambrosia</i> spp. | Boll weevil <i>Anthonomus grandis</i> | <i>Eurytoma tylodermatis</i> (Eurytomidae) |
| Cruciferous crops | Quick-flowering mustard <i>Arabidopsis thaliana</i> | Cabbageworms <i>Pieris</i> spp. | <i>Apanteles glomeratus</i> (Braconidae) |
| Grape | Blackberry <i>Rubus</i> spp. | Grape leafhopper <i>Erythroneura elegantula</i> | <i>Anagrus epos</i> (Mymaridae) |
| Grape | Bramble <i>Rubus ulmi folius</i> Elm <i>Ulmus minor</i> | Green grape leafhopper <i>Empoasca vitis</i> | <i>Anagrus</i> spp. (Mymaridae) |
| Lettuce | Flowering mixtures | <i>Liriomyza huidobrensis</i> | Eulophinae (idiobionts parasitic wasps) |
| Maize | Complex landscape | Armyworm <i>Pesudaletia unipuncta</i> | <i>Glyptapanteles militaris</i> <i>Meteorus communis</i> (Braconidae) |
| Maize | Wooded hedges and field margins | European corn borer <i>Ostrinia nubilalis</i> | <i>Eriborus terebrans</i> (Ichneumonidae) |
| Maize | Italian ryegrass <i>Lolium multiflorum</i> | European corn borer <i>Ostrinia nubilalis</i> | <i>Trichogramma brassicae</i> (Trichogrammatidae) |
| Maize | Giant ragweed <i>Ambrosia</i> spp. | European corn borer <i>Ostrinia nubilalis</i> | <i>Lydella grisescens</i> (Tachinidae) |
| Orchards | <i>Phacelia tanacetifolia</i> | San Jose scale <i>Quadraspidiotus perniciosus</i> | <i>Aphytis proclia</i> (Aphelinidae) |
| Peach | Ragweed <i>Ambrosia</i> spp. | Oriental fruit moth <i>Cydia molesta</i> | <i>Macrocentrus</i> spp. (Braconidae) |
| Sorghum | Sunflower <i>Helianthus annuus</i> | <i>Schizaphis graminum</i> | <i>Lysiphlebus testaceipes</i> (Braconidae) |
| Sugar cane | <i>Euphorbia</i> spp. | <i>Rhabdoscelus obscurus</i> | <i>Lixophaga sphenophori</i> (Tachinidae) |
| Sugar cane | <i>Borreria verticillata</i> <i>Hyptis atrorubens</i> | Cricket <i>Scapteriscus vicinus</i> | <i>Larra americana</i> (Sphecidae) |
| Sweet potato | Morning glory <i>Ipomoea asarifolia</i> | <i>Chelymorpha cassidea</i> | <i>Emersonella niveipes</i> (Eulophidae) |
| Vegetables crops | Wild carrot <i>Daucus carota</i> | <i>Popillia japonica</i> | <i>Tiphia popilliavora</i> (Tiphidae) |

Modified by authors from references listed at the end of this article.

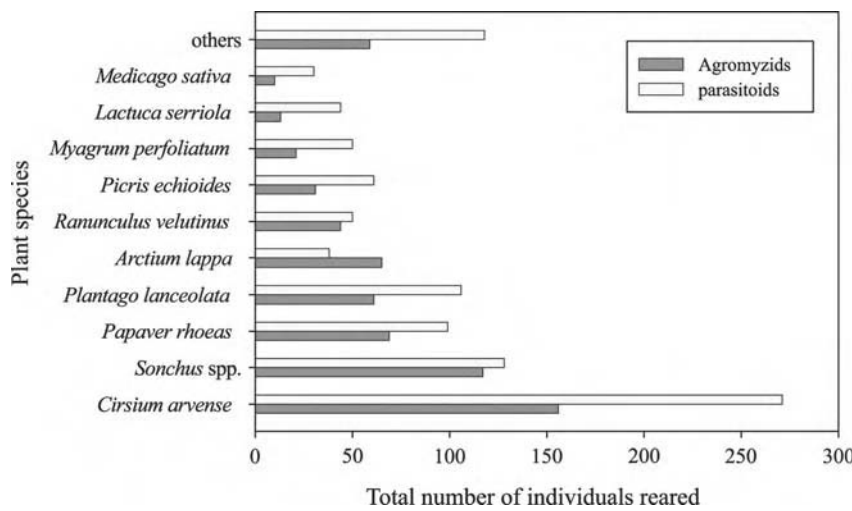


Fig. 1 Total number of agromyzid and parasitoid individuals that emerged from a mined foliage of weeds in a 2-year survey in northern Italy ecological compensation areas.

(*Salix* spp.), poplar (*Populus* spp.), hawthorn (*Crataegus monogyna*), elder (*Sambucus* spp.), and spindle tree (*Euonymus europaea*) proved to be suitable host plants for parasitoid multiplication.

Surveys of weeds near hedgerows in ecological compensation areas demonstrated the role of field margins as reservoirs of leafminer parasitoids. A total of 24 agromyzids species, and about 60 parasitoid species belonging to five families (Eulophidae, Braconidae, Tetracampidae, Eucoilidae, and Pteromalidae), were reared from mined foliage of 34 plant species.^[1,7] The agromyzids sampled in this study do not cause appreciable yield losses on European open field vegetables, and are valuable alternative hosts for many beneficials. Fig. 1 reports the contribution of weed species in supporting leafminer/parasitoid communities.

Parasitization can be affected also by microclimate changing due to the influence of non-crop plants [e.g., by reducing the temperature of the soil surface by interplanting Italian ryegrass (*Lolium multiflorum*) in seed maize fields, the survival of the released *Trichogramma brassicae* was increased].^[6]

CRITERIA AND PRACTICAL INTERVENTIONS TO ENHANCE BIOLOGICAL CONTROL AGAINST ARTHROPOD PESTS

The final decision in selecting plant species for the enhancement of biocontrol will have to consider characteristics of plant (annual/perennial; weed potential; floral architecture; pollen/nectar quality; quantity and nectar flow; suitability for herbivore arthropods), pest (host range, dispersal rate), and parasitoid (mouthpart morphology and body size, aggregative numerical response, dispersal rate). A better knowledge of which part of the flower is used by parasitoids

will help the selection of the right flowering plants to enhance parasitoid efficacy.^[1]

An important aspect in determining the suitability of a flower as a food source is the fit of floral architecture and insect mouthpart structure because the parasitoid needs to be able to access the floral nectars. Some authors suggest the term “selective food plant” for plants that selectively fulfill the needs of the beneficials without promoting the pest species. Selectivity can also be achieved through nectar composition.^[1]

There is empirical support that consumption of non-host foods leads to an increase in parasitism rates and to a decrease in host densities; several studies in laboratories and fields have demonstrated higher rates of parasitism when nonhost resources are available compared to when they are less available or absent.^[1]

Many examples are reported on practical interventions carried out at farm level to increase the efficiency of parasitoids in biological control.^[2-5] Only recently has the attention of researchers focused on management at landscape level.^[1]

Practical ways at farm level to enhance conservation biological control by parasitoids against insect pests are as follows:

1. Rational management of field margin vegetation, including wildflowers and hedgerows.^[2]
2. Sowing annual flowering mixture strips.^[1,5,6]

For example, in the UK field margin, diversification is used in agri-environment schemes to enhance aphid natural enemies, including parasitoids, on winter cereal fields,^[1] the results illustrate the importance of early parasitoid activity for initiating conservation biological control against aphid pests.

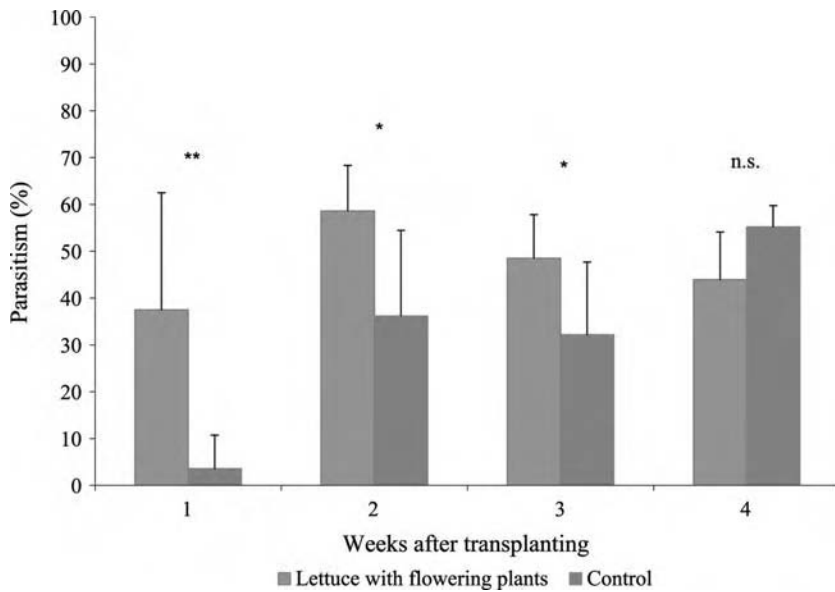


Fig. 2 Percent parasitism of *Liriomyza huidobrensis* (mean \pm SD) in lettuce plots adjacent to flowering strips as a function of weeks from transplanting (χ^2 test, $**P < 0.01$, $*P < 0.05$, n.s., $P > 0.05$).

An example of the effect of flowering mixture strips in enhancing the percentage of parasitization against agromyzid pests was demonstrated on lettuce crop (Fig. 2).^[1]

Besides the effects on the parasitism determined by factors at farm scale, also the habitat diversity and connectivity in the landscapes are very important in improving parasitoid populations.^[8,9] There is increasing evidence that habitat fragmentation can disrupt host–parasitoid relationships. Recently, a scale dependency of landscape effects on parasitism was demonstrated.^[1] The author found that the parasitism rate of the agromyzid *Melanagromyza aeneoventris* was affected by both habitat type and landscape structure; the highest parasitization was found on fallow habitats, where the parasitism rate was about 50%. Herbivores will suffer more from parasitism in landscapes characterized by a high proportion of structurally rich landscapes and a high proportion of large and undisturbed habitats.^[1]

In a study on temporal variation in the response of parasitoids to agricultural structure, it was pointed out that the effects of landscape structure on parasitism are not adequately characterized by short-term studies.^[10] The authors found that agricultural landscape structure influenced the temporal dynamics of armyworm larvae (*Pseudaletia unipuncta*) parasitism. While the parasitoid *Glyptapanteles militaris* (Braconidae) was equally present in the simple and complex landscape, another braconid, *Meteorus communis*, was found mostly in the complex landscape. Overall, percentage parasitism differed between landscapes from year to year with different trends in the simple and complex landscapes.

CONCLUSION

Natural vegetation is very important for the life cycle and activity of parasitoids; rational management of noncrop plants within farming systems becomes crucial in improving biological control on crops. The criteria for the selection of the plant to be used are complex and range from ecological to agronomic considerations. Many cases of interventions are reported at small scale (field and/or farm); however, there is an increasing body of evidence that habitat fragmentation and landscape structure can effect the efficiency of beneficial arthropods. Thus there is the need to develop these studies at landscape scale.

REFERENCES

1. *Landscape Management of Functional Biodiversity*, Proceedings of the 1st Meeting of the Study Group, Bologna, Italy, May 11–14, 2003; Rossing, W.A.H., Poehling, H.-M., Burgio, G., Eds.; IOBC/WPRS, INRA-Centre de Recherches de Dijon: France, IOBC/WPRS Bulletin, 2003; 26 (4), 83–88; 117–134; 197–220.
2. Altieri, M.A.; Letourneau, D.K. Vegetation management and biological control in agroecosystems. *Crop Prot.* **1982**, 1 (4), 405–430.
3. van Emden, H.F. Plant diversity and natural enemy efficiency in agroecosystems. In *Critical Issues in Biological Control*; Mackauer, M., Ehler, L.E., Roland, J., Eds.; Intercept Ltd.: Andover, UK, 1990; 63–80.
4. Andow, D.A. Vegetational diversity and arthropod population response. *Annu. Rev. Entomol.* **1991**, 20, 1228–1235.

5. Delucchi, V. Una nuova frontiera: la gestione ambientale come prevenzione. In *Atti Giornata Sulle Strategie Bio-Ecologiche di Lotta Contro Gli Organismi Nocivi, Sassari, Italy, April 11, 1997*; Prota, R., Pantaleoni, R.A., Eds.; CNR: Sassari, Italy, 1997; 35–57.
6. Landis, D.A.; Wratten, S.D.; Gurr, G.M. Habitat management to conserve natural enemies of arthropod pests in agriculture. *Annu. Rev. Entomol.* **2000**, *45*, 175–201.
7. Burgio, G.; Lanzoni, A.; Accinelli, G.; Masetti, A.; Navone, P. The Role of Weeds as Reservoirs of Parasitoids of Agromyzidae. In *Antonie van Leeuwenhoek Symposium, Proceedings of the 7th European Workshop on Insect Parasitoids*, Harlem, The Netherlands, October 1–6, 2000; Laboratory of Entomology: Wageningen, The Netherlands, p. 47.
8. Marino, P.C.; Landis, D.A. Effect of landscape structure on parasitoid diversity and parasitism in agroecosystems. *Ecol. Appl.* **1996**, *6* (1), 276–284.
9. Landis, D.A.; Haas, M.J. Influence of landscape structure on abundance and within-field distribution of European corn borer (Lepidoptera: Pyralidae) larval parasitoids in Michigan. *Environ. Entomol.* **1992**, *21* (2), 409–416.
10. Menalled, F.D.; Costamagna, A.C.; Marino, P.C.; Landis, D.A. Temporal variation on the response of parasitoids to agricultural landscape structure. *Agric. Ecosyst. Environ.* **2003**, *96*, 29–35.

Nematicides

Stephen R. Koenning

*Department of Plant Pathology, College of Agriculture and Life Sciences,
North Carolina State University, Raleigh, North Carolina, U.S.A.*

INTRODUCTION

Nematodes are microscopic aquatic roundworms found in virtually all environments. Different groups of nematodes occupy various ecological niches in nature. Most nematodes feed on bacteria, fungi, algae, and even other nematodes; but some are specialized parasites of animals or plants. Virtually all vascular plants are susceptible to at least one species of plant-parasitic nematode. Pesticides used to control or manage plant-parasitic nematodes are referred to as nematicides, whereas chemical compounds used to control animal-parasitic nematodes are called anthelmintic agents. Nematicides are used extensively on a number of high-value crops such as strawberries, tobacco, banana, turf, and pineapple, but are generally too costly for use on low value crops such as soybean or wheat.

PLANT-PARASITIC NEMATODES

Most nematode parasites of plants feed on or in the root system of the plant, although a few nematode species also infect leaves and stems. The most damaging plant-parasitic nematodes are obligate parasites of plants. All plant-parasitic nematodes feed on host tissue through a hollow, needle-like structure referred to as a stylet. Depending on the genus of nematode involved, nematodes may feed on the outside of roots (ectoparasites) or internally (endoparasites). Many taxonomic classes of plant-parasitic nematodes stimulate the plant to develop specialized nurse cells that supply nutrients to the nematode for growth, development, and reproduction. Nematode life cycles may be quite diverse and specialized, but generally have a typical pattern from egg to adult. A single-celled egg develops to a first stage juvenile (j1), which molts in the egg to become a second stage juvenile (j2). The second stage juvenile hatches from the egg and continues to develop through two more stages (j3 and j4) to become a sexually mature adult. The nematode must feed to develop beyond the j2 stage. Nematode taxon vary as to which stage (egg, juvenile in an egg, j3, j4, or adult) is the survival stage.

Although most nematodes are found in soil, they are basically aquatic organisms that move and live in

the soil water. Nematodes may be found in any soil type, but the population densities of these organisms tend to be greatest in coarse textured (sandy) soil as is the associated plant damage. Nematicides must be water-soluble to effectively impact nematode populations. Since many crops susceptible to nematodes are grown on well-drained soil, the potential for nematicides to move into ground or surface waters is high.

Nematode damage to plants is often insidious and difficult to detect. The root system may be debilitated, limiting the availability of water and nutrients to the plant, thus resulting in a general stunting. The specialized nurse cells induced by some species of plant-parasitic nematodes cause photosynthate to be accumulated for nematode nutrition rather than plant biomass. In the case of perennial plants, premature death or a decline in productivity may be the only above ground symptom. Plant-parasitic nematodes often predispose plants to diseases caused by fungi or bacteria, and many vector plant viruses.

NEMATODE MANAGEMENT

Relative to other plant pathogens, nematodes are easy to quantify per unit of soil. Diagnostic labs are available in many areas to determine nematode species and population densities based on quantitative assays. The decision to use a nematicide or other management tactic such as a resistant variety, sanitation, or rotation should be based on the damage potential and population density of the nematode species in question, the crop value, and potential returns. Generally, an integrated pest management approach that combines several tactics, which may include chemical control, is needed to obtain adequate crop yields in the presence of damaging numbers of these pathogens.

TYPES OF NEMATICIDES

The majority of nematicides can be classified as either fumigants or non-fumigants (Table 1). In general, fumigant nematicides must be applied prior to planting because of their phytotoxicity. Specialized equipment is required to inject and seal fumigants in the soil. Fumigant nematicides, with the exception of

Table 1 Common names, chemical class, U.S. regulatory status, and toxicity class of selected nematicides

| Common name | Chemical class ^a | Status/toxicity class ^b |
|-----------------------------|-----------------------------|------------------------------------|
| Carbon disulfide | — | Obsolete, II |
| Chloropicrin | HHC | RUP, I |
| 1,3 Dichloropropene | HHC | RUP, II |
| Methyl bromide | HHC | RUP (Banned—2005), I |
| Ethylene dibromide (EDB) | HHC | Banned, I |
| Dibromochloropropane (DBCP) | HHC | Banned, II |
| Metam-sodium | MIT | RUP, I |
| Aldicarb | C | RUP, I |
| Oxamyl | C | RUP, I |
| Fenamiphos | OP | RUP, I |
| Terbufos | OP | RUP, I |
| DiTera | B | III |
| Clandosan | B | III |

^aChemical classes: HHC is halogenated hydrocarbon; MIT is a methyl isothiocyanate liberator; C is a carbamate; OP is for organophosphate; B indicates a biological material.

^bRUP indicates a restricted use pesticide in the United States. Toxicity class: I is very toxic, and IV is lowest toxicity.

metam-sodium and carbon disulfide, are halogenated hydrocarbons. Non-fumigant chemical nematicides are classified as either carbamates or organophosphate pesticides. The carbamate and organophosphate nematicides tend to be systemic in the plant and since they affect animal nervous systems, they also are effective insecticides. In recent years, biologically based products for nematode control have been introduced to the market, but the efficacy and usefulness of these products have not been fully evaluated.

Fumigant Nematicides

The first materials developed as nematicides were various volatile gases (fumigants) that were often phytotoxic, and thus limited to preplant application. Carbon disulfide was used as an aid in controlling phyloxera of grape in the 1800s and was subsequently shown to be useful in controlling nematodes. The first halogenated hydrocarbon to be tested and used as a nematicide was chloropicrin (“tear gas”) in the early 1900s. Subsequently, methyl bromide was shown to be an effective general biocide that was also effective against nematodes. Methyl bromide, however, must be applied and covered with a tarp or plastic because of its extreme volatility. The fumigant DD (1,3-dichloropropene; 1,2-dichloropropane) was discovered by Carter in 1943. Fumigation to control nematodes came into wide-scale use after WWII with the commercial production of DD and ethylene dibromide (EDB). Soil fumigation with these materials convincingly demonstrated the yield-limiting effects of nematodes on crops and contributed to increased interest in nematodes as

the causal agents of a number of important diseases. Dibromochloropropane (DBCP) was registered for at-planting use on a number of crops because of its low phytotoxicity. Subsequently, DBCP was widely used in perennial and orchard crops until its ban in 1980. Metam-sodium and related materials liberate cyanide upon addition to the soil and are thus toxic to many organisms including nematodes, fungi, and weed seeds.

Non-Fumigant Nematicides

Nonvolatile liquid and granular pesticides in the carbamate or organophosphate chemistry classes have nematicidal activity and are labeled as “insecticide/nematicides” (Table 1). Aldicarb, carbofuran, and oxamyl are examples of nematicides in the carbamate class; while fenamiphos, ethoprop, and terbufos are organophosphates. Both carbamates and organophosphates act as cholinesterase (nerve synapse) inhibitors and tend to have systemic activity within the plant. A major drawback for all of these materials is their high mammalian toxicity. Although nonfumigant nematicides are generally less effective for nematode control than fumigant nematicides, they are widely used because of the ease of application (pre- or postplanting application), low phytotoxicity, and systemic insecticidal activity.

Other Nematicidal Materials

Much interest has been expressed in biological and other materials for nematode control because of the

acute toxicity of most nematicides and the potential for groundwater contamination. DiTerra is a fermentation product made from the killed fungus *Myrothecium verrucaria*. Clandosan, a chitin-based product derived from the exoskeletons of shellfish augmented with “organic” sources of urea, is marketed as a nematicide. Urea degrades into ammonia, which has nematicidal properties. Ammonia, however, when applied at rates high enough to be nematicidal is usually phytotoxic. A number of waste products and other materials, generally with a low carbon to nitrogen ratio, have been exploited for their nematicidal properties. Various rotation crops, green manures, and poultry litter have also shown nematicidal activity. A natural product that has received much attention are extracts and portions of the neem tree *Azadirachta indica*.

In the past two decades, a number of anthelmintic compounds have been employed in human and veterinary medicine to manage animal-parasitic nematodes. Some of these compounds may have promise in agricultural nematology. Avermectins, produced by species of *Streptomyces avermitilis*, in particular, have shown promise in controlling nematodes at very low rates. Currently, the price of these compounds limits their potential as agricultural nematicides. Certain endotoxins produced by strains of the bacteria *Bacillus thuringiensis* also have shown nematicidal activity.

FUTURE CONCERNS

The registrations of many nematicides have been justifiably cancelled for environmental and human health considerations. Few new nematicides for agricultural use have been developed in the past two decades, despite a pressing need for these pesticides. There are

several reasons for the lack of development of nematicides: 1) soil application of nematicides, which must necessarily be water-soluble, makes them subject to leaching; 2) chemical companies are unwilling to accept the risk associated with the marketing of new products with high mammalian toxicity; and 3) the market for nematicides is perceived as being small relative to the cost of pesticide registration. The problem is further exacerbated by the fact that although resistance to nematicides has not been a problem thus far, microbial degradation of some of these products has become an issue where these compounds have been used repeatedly.

BIBLIOGRAPHY

- Barker, K.R., Pederson, G.A., Windham, G.L., Eds.; *Plant and Nematode Interactions*; American Society of Agronomy, Crop Science Society of America, and Soil Science Society of America: Madison, WI, 1998.
- D'Abbaso, T. The nematicidal effect of organic amendments: a review of the literature, 1982–1984. *Nematol. Mediterr.* **1995**, *23*, 299–305.
- Johnson, A.W.; Feldmesser, J. Nematicides—a historical perspective. In *Vistas on Nematology*; Veech, J.A., Dickson, D.W., Eds.; Society of Nematologists: Hyattsville, MD, 1987; 448–454.
- Koening, S.R.; Overstreet, C.; Noling, J.W.; Donald, P.A.; Becker, J.O.; Fortnum, B.A. Survey of crop losses in response to phytoparasitic nematodes in the United States for 1994. *Suppl. J. Nematol.* **1999**, *31* (4S), 587–618.
- Ristaino, J.B.; Thomas, W. Agriculture, methyl bromide, and the ozone hole—Can we fill the gaps? *Plant Dis.* **1997**, *81*, 964–977.
- <http://www.ianr.unl.edu/son/> (Society of Nematologists Home Page).

Neurological Effects of Insecticides

Michael E. Scharf

Department of Entomology, Purdue University, West Lafayette, Indiana, U.S.A.

INTRODUCTION

The vast majority of pesticides with neurological effects are those targeted at insects, arthropods, and nematodes. For convenience, these chemicals will be referred to here as insecticides. The neurotoxic insecticides are capable of exerting a broad range of effects on insects, arthropods, and nematodes that ultimately result in mortality. In most basic terms, the neurological effects of insecticides (or any pesticide) are categorized as either neuroexcitatory or neuroinhibitory (see Fig. 1). At the whole organism level, the observable behavioral effects of neuroexcitation are hyperactivity, tremors, and rigid paralysis, while neuroinhibition results in immobility and flaccid paralysis. The effects of neuroexcitation generally occur rapidly, while those of neuroinhibition take longer to become apparent. The actual causes of mortality in insects exposed to neurotoxicants are less understood. In the case of neuroexcitation, mortality is apparently caused by energy depletion and neuromuscular fatigue. In the case of neuroinhibition, the potential causes of mortality are not as apparent but possibly relate to oxygen deprivation and/or reduced respiratory capacity.

NERVOUS SYSTEM FUNCTION AND TERMINOLOGY

Electrical impulses called action potentials travel along neurons and provide the basis of nervous system function (Fig. 2A). The action potentials are waves of electrical energy that are perpetuated by sodium and potassium ions entering and exiting neurons (respectively) through channels specific to each ion. These channels are sometimes referred to as “voltage-gated,” because they only function under specific voltage- or charge-dependent conditions. Gaps between neurons are called synapses. Action potentials traveling through the nervous system are carried across synapses by chemical messengers called neurotransmitters (Fig. 2B–2D). Neurotransmitter release into a synapse is triggered by the arrival of an action potential. Examples of neurotransmitters are acetylcholine, gamma amino butyric acid (GABA), and glutamate. Acetylcholine is a neurotransmitter that activates sodium channels in postsynaptic neurons and results

in the initiation of new action potentials (Fig. 2C). Because it stimulates the production of new action potentials, acetylcholine is categorized as an “excitatory” neurotransmitter. On the other hand, because they elicit an influx of negatively charged chloride ions into postsynaptic neurons, GABA and glutamate are referred to as “inhibitory” neurotransmitters (Fig. 2D). After neurotransmitters traverse synapses, they bind to receptors and either 1) cause depolarizing receptor potentials that, in turn, elicit the formation of action potentials in the postsynaptic neuron (as with acetylcholine and the excitatory neurotransmitters; Fig. 2C); or 2) cause intracellular hyperpolarization or an “inhibitory potential” that counteracts the influence of excitatory impulses (as with GABA, glutamate, and other inhibitory neurotransmitters; Fig. 2D).

NEUROLOGICAL INSECTICIDE TARGET SITES

Insecticide target sites are defined as the specific biochemical or physiological sites within an organism that insecticide molecules interact with to create toxic effects. The physical properties of insecticides dictate the target sites that they are capable of interaction with. There are several highly relevant neurological target sites that are acted upon by insecticides (see Tables 1 and 2). This summary focuses on target sites of the most prevalent neurotoxic insecticides in use worldwide. The neurological insecticide target sites reviewed here include the acetylcholinesterase enzyme, voltage-gated sodium channels, GABA- and glutamate-gated chloride channels, and nicotinic acetylcholine receptors. The actions of insecticides at these sites are diverse and range from enzyme inhibition, to receptor agonism (stimulation), receptor antagonism (blockage), and ion channel modulation (altered gating kinetics).

Acetylcholinesterase Enzyme

Acetylcholinesterase is an enzyme that occurs in the central nervous system. It functions by removing acetylcholine from its postsynaptic receptor. The result of this action is the hydrolysis of acetylcholine into acetate and choline and, ultimately, the initiation of action potentials at precise, exact intervals. Organophosphate and carbamate insecticides inhibit the

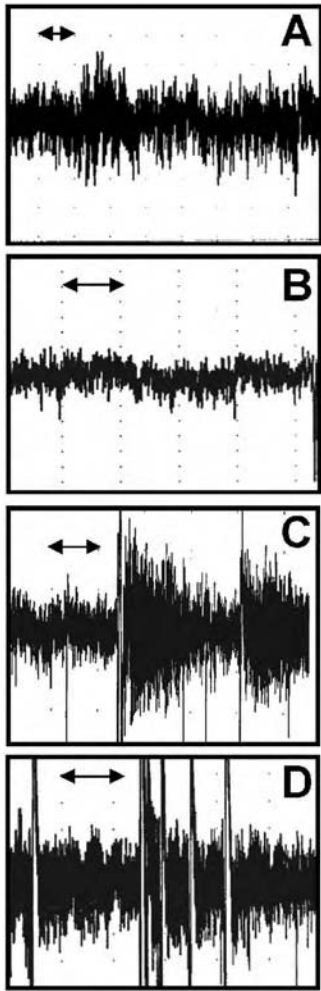


Fig. 1 Spontaneous bursting activity from intact ventral nerve cords of the American cockroach *Periplaneta americana*, L., obtained using an extracellular suction recording electrode. The distance between arrowheads (\leftrightarrow) indicates 10 sec of recording time. All recordings took place in physiological saline at room temperature. Recordings shown in the figure are: (A) baseline activity; (B) depressed activity in the presence of 10 mM of the inhibitory neurotransmitter GABA; (C) repetitive bursts of excitatory activity in the presence of 10 μ M of the type I pyrethroid permethrin (active at voltage-gated sodium channels); and (D) random excitatory bursts in the presence of 10 mM GABA and 10 μ M of the phenylpyrazole insecticide fipronil (active at GABA-gated chloride channels).

acetylcholinesterase enzyme, which results in the prolonged binding of acetylcholine to its postsynaptic receptor. Ultimately, these actions lead to the death of an organism from prolonged neuroexcitation. Organophosphate insecticides are generally very long or even irreversible inhibitors of acetylcholinesterase. Organophosphates must be enzymatically activated to their oxon metabolites by endogenous oxidase enzymes (the cytochromes P450) before they can serve as effective acetylcholinesterase inhibitors. Unlike the

organophosphates, carbamate insecticides are fast but reversible acetylcholinesterase inhibitors. They do not require oxidative activation to elicit toxicity (although some can be oxidatively metabolized to even more potent acetylcholinesterase inhibitors). Both organophosphate and carbamate insecticides possess relatively high mammalian toxicity; however, the carbamates are generally more *hazardous* because of the greater affinity they possess for the acetylcholinesterase enzyme (*hazard* is a toxicological term that is defined as the interaction of toxicity and exposure; i.e., hazard = toxicity \times exposure).

Voltage-Gated Sodium Channels

Voltage-gated sodium channels are responsible for the initiation and perpetuation of action and receptor potentials in neurons, both central and peripheral. Insecticides that act upon sodium channels include DDT (and related analogs), natural pyrethrins, synthetic pyrethroids, and dihydropyrazoles. Sodium channels are gated (i.e., activated and inactivated) at highly precise intervals based on specific physiological properties. DDT, pyrethrins, and pyrethroids act by modulating sodium channels, which results in altered gating kinetics. Generally, DDT, pyrethrins, and pyrethroids affect sodium channels by causing 1) activation at lower thresholds or 2) inactivation later than would occur under normal circumstances. The end result is prolonged flow of sodium currents into neurons and neuronal dysfunction because of excessive neuroexcitation. Recent research into the molecular mechanisms of sodium channel function and pharmacology suggests that DDT, pyrethrin, and pyrethroid insecticides alter the kinetics of gating through allosteric modulation of the channel, rather than by serving as true agonists as originally suspected.

The dihydropyrazoles are a relatively new class of insecticides that act as sodium channel antagonists. Dihydropyrazole effects make it appear as if an organism is paralyzed. At the neuronal level, dihydropyrazole toxicity appears to be a result of sodium current blockage. This effect is in great contrast to the gate-modifying sodium channel toxins noted above and suggests that the molecular mode of action on sodium channels by dihydropyrazoles involves interaction with the channel pore itself.

Gamma Amino Butyric Acid and Glutamate-Gated Chloride Channels

Gamma amino butyric acid and glutamate are inhibitory neurotransmitters that elicit the influx of chloride ions into central neurons through chloride channels.

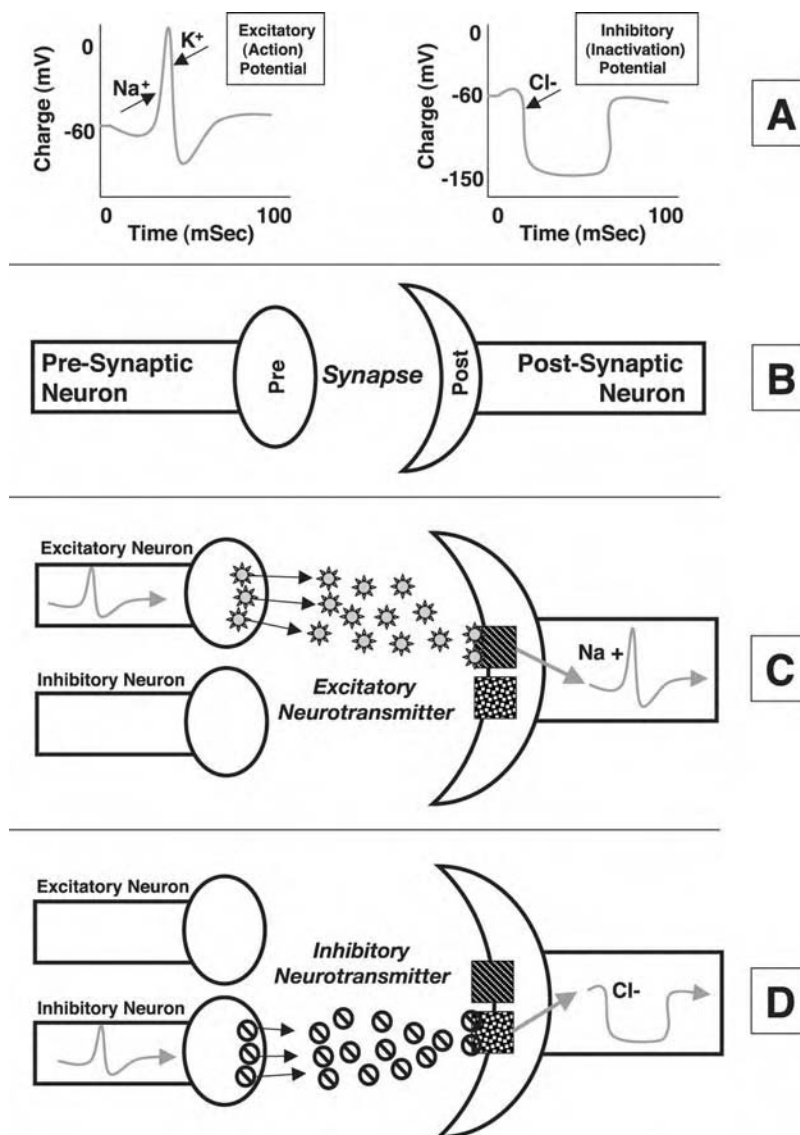


Fig. 2 Illustrations depicting nervous system structure, function, and associated terminology. (A) Changes in charge over time (in milliseconds; msec) inside neurons, in association with excitatory “action” potentials (left) and inhibitory “inactivation” potentials (right). Action potentials are associated with an influx of positively charged sodium (Na^+), then potassium (K^+) out of neurons, and a resulting increase in net charge above 0 mV from resting levels of approximately -60 mV. Inactivation potentials are associated with the flow of negatively charged chlorine (Cl^-) into neurons, and a resulting increase in net negative charge within neurons. (B) Diagram of a synapse, which is a gap that separates two neurons (i.e., pre- and postsynaptic neurons). (C) Excitatory neurotransmitter release is stimulated by the arrival of an action potential at the synapse of an excitatory neuron. Excitatory neurotransmitters (\star) bind specific receptors on postsynaptic neurons (hatched box), which stimulates the production of new action potentials. (D) Inhibitory neurotransmitter release is stimulated by the arrival of an action potential at the synapse of an inhibitory neuron. Inhibitory neurotransmitters (\ominus) bind specific receptors on postsynaptic neurons (checkered box), which elicits the production of inhibitory, inactivation potentials.

Insecticides that are active at chloride channels produce either neuroexcitation or neuroinhibition. Early insecticides such as cyclodienes and polychlorocycloalkanes, as well as the newer phenylpyrazoles, function as antagonists at the GABA-gated chloride channel complex. In the case of chloride channels, antagonism results in a blockage of neuroinhibitory chloride currents, leading ultimately to neuroexcitation.

In arthropods, glutamate-gated chloride channels occur at skeletal neuromuscular junctions of both the peripheral and central nervous systems. Avermectin and milbemycin insecticides act at glutamate-gated chloride channels by producing effects that are opposite to insecticides that act at GABA-gated chloride channels. Because they function as agonists of chloride channels, avermectins and milbemycins elicit

Table 1 Target sites and effects of neurotoxic insecticides in wide-scale use

| Neurological target site | Insecticide/insecticide class | Mode of action | Generalized neurological effect |
|----------------------------------|-----------------------------------|--------------------------|-------------------------------------|
| Acetylcholinesterase enzyme | organophosphate | inhibition | excitation |
| | carbamate | inhibition | excitation |
| Voltage-gated sodium channel | DDT and analogs | modified gating kinetics | excitation |
| | pyrethrin/pyrethroid | modified gating kinetics | excitation |
| | dihydropyrazole | antagonism | inhibition |
| GABA-gated chloride channel | cyclodiene, polychlorocycloalkane | antagonism | excitation |
| | phenylpyrazole | antagonism | excitation |
| Glutamate-gated chloride channel | avermectin, milbemycin | agonism | inhibition |
| Nicotinic acetylcholine receptor | neonicotinoid/chloronicotinyl | agonism | excitation, inhibition ^a |
| | spinosyn/spinosoid ^b | agonism | inhibition |

^aNeonicotinoids initially cause excitation, followed by inhibition.

^bEffects have also been noted at the GABA-gated chloride channel.

increased chloride current flow into neurons. This increased chloride current results in intracellular hyperpolarization and neuroinhibition via the cancellation of positively charged excitatory impulses (carried by sodium currents).

Nicotinic Acetylcholine Receptor

Nicotinic acetylcholine receptors occur in postsynaptic membranes of the arthropod central nervous system. They are termed “nicotinic” because they are bound with great affinity by the plant-derived insecticide nicotine. There are also other classes of acetylcholine receptors (i.e., muscarinic receptors) that are more resistant to the effects of nicotinoid insecticides

currently in use, and which are greatly outnumbered in the arthropod nervous system by nicotinic receptors. The excitatory neurotransmitter acetylcholine functions by binding to the postsynaptic acetylcholine receptor, causing an influx of sodium ions and the formation of action potentials. Nicotinic acetylcholine receptors are the target sites for neonicotinoid/chloronicotinyl and spinosyn/spinosoid insecticides that are currently in wide-scale use. Experimental evidence suggests that these two types of insecticides mainly cause nervous disruption by acting as agonists at nicotinic acetylcholine receptors. The primary toxic symptom associated with the nicotinic acetylcholine receptor agonists is neuroexcitation; however, longer-term neuroinhibitory effects have been observed. While the

Table 2 Examples of insecticides (by common name) that occur in various neurotoxic insecticide classes

| Insecticide class | Insecticides |
|--|--|
| Avermectin ^a | abamectin, ivermectin, doramectin |
| Carbamate | aldicarb, bendiocarb, carbaryl, carbofuran, methiocarb, propoxur |
| DDT and analogs | DDT, dicofol, kelthane, methoxychlor, ovex |
| Cyclodiene/polychlorocycloalkane | aldrin, chlordane, dieldrin, endosulfan, heptachlor, lindane, toxaphene |
| Dihydropyrazole | indoxacarb |
| Milbemycin ^a | milbemycin, moxidectin |
| Neonicotinoid/chloronicotinyl ^b | acetamiprid, clothianidin, dinotefuran, imidacloprid, nitenpyram, nithiazine, thiacloprid, thiamethoxam |
| Organophosphate | azinphos, chlorpyrifos, diazinon, dimethoate, ethyl-parathion, fenitrothion, fonofos, malathion, methyl-parathion, pirimiphos, propetamphos, phorate, temephos, terbufos |
| Phenylpyrazole | fipronil, fipronil-sulfone |
| Pyrethrin | natural pyrethrins, chrysanthemum extract |
| Pyrethroid | allethrin, cypermethrin, fenvalerate, permethrin, resmethrin |
| Spinosyn/spinosoid | spinosid, spinosyn A, spinosyn B, macrocyclic lactone |

^aSee Ref.^[4].

^bSee Ref.^[6].

nature of this late neuroinhibition is unclear, it has also been observed with nicotine, suggesting a common symptom of poisoning at this target site. Some observations also suggest activity at the GABA receptor, as is the case with the spinosyns.

CONCLUSION

As highlighted here, there are numerous classifications of the interactions between insecticides and their neurological target sites. These interactions include hydrolytic enzyme inhibition, modulation of ion channel gating kinetics, and agonism or antagonism at complexes of neurotransmitter receptors and ion channels. Given these relatively diverse interactions, there are only two end results that are observable in affected neurons: neuroexcitation and neuroinhibition. Even as new types of neurotoxic pesticides with new sites of action evolve or are discovered, these fundamental effects will likely remain as the basis of pesticide-induced neuronal dysfunction.

ACKNOWLEDGMENTS

I acknowledge the support of Blair Siegfried (University of Nebraska-Lincoln), in whose lab I was working when I made the *P. americana* recordings. This is

publication No. 16775 of the Purdue University Agricultural Experiment Station, West Lafayette, IN, USA.

REFERENCES

1. Matsumura, F. *Toxicology of Insecticides*; Plenum: New York, 1986.
2. Sparks, T.C.; Crouse, G.D.; Durst, G. Natural products as insecticides: the biology, biochemistry and quantitative structure activity relationships of spinosyns and spinosoids. *Pest Manag. Sci.* **2001**, *57* (10), 896–905.
3. Salgado, V.L. The modes of action of spinosad and other insect control products. *Down to Earth News* **1997**, *52* (1), 35–43.
4. Bloomquist, J.R. GABA and glutamate receptors as biochemical sites for insecticide action. In *Biochemical Sites of Insecticide Action and Resistance*; Ishaaya, I., Ed.; Springer-Verlag: Berlin, 2001; 17–41.
5. Zlotkin, E. Insecticides affecting voltage-gated ion channels. In *Biochemical Sites of Insecticide Action and Resistance*; Ishaaya, I., Ed.; Springer-Verlag: Berlin, 2001; 43–76.
6. Nauen, R.; Ebbinghaus-Kintscher, U.; Elbert, A.; Jeschke, P.; Tietjen, K. Acetylcholine receptors as sites for developing neonicotinoid insecticides. In *Biochemical Sites of Insecticide Action and Resistance*; Ishaaya, I., Ed.; Springer-Verlag: Berlin, 2001; 77–105.
7. Perry, A.S.; Yamamoto, I.; Ishaaya, I.; Perry, R.Y. *Insecticides in Agriculture and Environment*; Springer-Verlag: Berlin, 1998.

Non-Indigenous Species: Crops and Livestock

David Pimentel
Anne Wilson

*Department of Entomology, College of Agriculture and Life Sciences, Cornell University,
Ithaca, New York, U.S.A.*

INTRODUCTION

Approximately 50,000 non-indigenous (non-native) species are estimated to have been introduced to the United States. Some of these species are beneficial; for example, 99% of U.S. food comes from introduced crops (e.g., wheat, corn, rice, and beans) and introduced livestock (e.g., cattle, poultry, and swine). The introduced species provide a value of approximately \$800 billion per year.^[1] Other exotic species have been introduced for landscape restoration, biological pest control, sport, pets, and food processing, and provide significant benefits. Some non-indigenous species, however, have caused major environmental and economic losses in agriculture, forestry, and several other segments of the U.S. economy.

Estimating the full extent of environmental damages caused by exotic species is extremely difficult. One estimate is that the U.S. exotics cause more than \$143 billion in damage and control costs per year. It is also estimated that the invasive species are responsible for about 40% of the extinctions in the United States. Invasive plants are probably the most serious causes of extinctions. Approximately 400 of the 958 species listed as threatened or endangered under the Endangered Species Act are considered to be at risk because of competition with, and predation and parasitism by, non-indigenous species.

WORLD FOOD SUPPLY

Malnourishment is caused by a lack of adequate food, as well as poor distribution of food. According to the World Health Organization, more than 3 billion people are malnourished.^[2,3] This is the largest number and proportion of malnourished people ever reported! In assessing malnutrition, WHO includes deficiencies of calories, protein, iron, iodine, and vitamin A, B, C, and D shortages in its evaluation.^[4,5] Humans die from shortages of any one or combinations of these nutrients. Also, malnourishment diminishes the ability of people to work and enjoy the pleasures of life.

Food, per capita, has been declining since 1984, based on available cereal grains according to the Food and Agriculture Organization^[6] (Fig. 1). This is alarming because cereal grains make up about 80% of the world's food supply. Although grain yields per hectare in both developed and developing countries are still increasing, the rate of growth is slowing, while the world population and its food needs escalate.^[6,7] Specifically, from 1950 to 1980 U.S. grain yields increased by about 3% per year, but since 1980 the annual rate of increase for corn and other major grains has declined to a yearly increase of only about 1%.^[8]

CROPS THAT PROVIDE 90% OF WORLD FOOD

Of the 250,000 species of plants in the world, only 15 species (only 0.006% of the plant species) provide more than 90% of the world's dietary energy supply (DES) for humans (Table 1). The cereal grains provide nearly 80% of the DES. However, it is known that humans consume or have consumed some 20,000 different species of plants.

The cereal grains provide most of the food for several reasons: 1) grains are highly productive under a wide range of soils and rainfall patterns; 2) grains have low moisture levels and thus can be transported easily compared with crops that have 80% to 90% moisture such as potatoes; and 3) grains store easily and for relatively long periods of time.

The legumes and root crops provide a significant quantity of the food in the world. In particular, the legumes are essential to those people living as vegetarians. The legumes are high in lysine, but they are low in methionine, which exists in relatively high levels in cereal grains. Thus people are able to obtain a suitable balance in amino acids by consuming an appropriate mix of cereal grains and legumes.

Root crops, such as potatoes, have a good mix of amino acids but have a high level of moisture, about 80%. This high moisture level makes potatoes costly to transport in contrast to grains that have 13% to 15% moisture. Potatoes also spoil in storage more easily than grains.

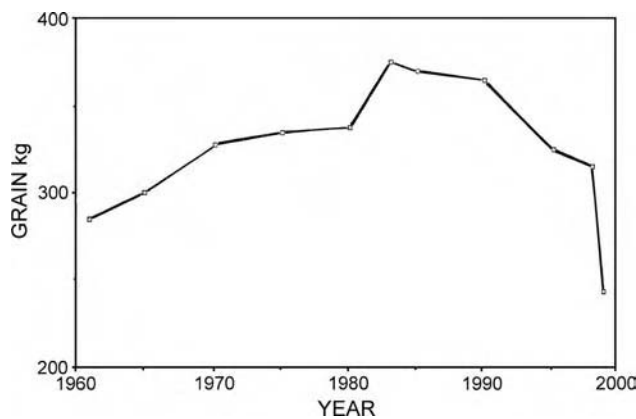


Fig. 1 Cereal Grain Production per capita in the world from 1961 to 2000 (FAO, 1961–2000, Quarterly Bulletin of Statistics. 1–13).

LIVESTOCK THAT PROVIDE 90% OF THE WORLD’S ANIMAL PROTEIN

Of the six livestock groups listed in Table 2 that provide about 90% of the animal protein consumed by

humans, five are mammal species. From the mammals we obtain both milk and meat. The five species of mammals are from a total of 4300 species of mammals in the world or about 0.1% of the mammal species.

The one group of livestock that are not mammals are the poultry. The chicken is one species out of about 9700 species of birds in the world. Chickens provide humans with both eggs and meat. In the United States, for example, more than 8 billion chickens are raised each year.

One interesting fact related to livestock production in the United States is that all the livestock outweigh the human population by about five times. Also, livestock consume more than 250 million tons of grain per year. This is enough grain to feed 850 million people as vegetarians.

Except for hogs and poultry, all the livestock species can be raised on only forage. Hogs and poultry require grain for their culture. Poultry in particular are relatively efficient in the conversion of grain into protein. It only takes about 2.5 kg of grain to produce 1 kg of chicken, whereas about 15 kg of grain is required to produce 1 kg of beef. If the beef were produced on forage, about 100 kg of forage would be necessary.

Table 1 Major crops that provide approximately 90% of the U.S. and World’s DES and their origins

| Crop | %DES | Source | Origin |
|--------------------|------|--------|---|
| Wheat | 23 | FAO | SW Asia (Syria, Jordan, Turkey) ^a |
| Rice | 26 | FAO | China (Middle Yangtze Basin) ^b |
| Maize | 7 | FAO | Mexico ^c |
| Potatoes | 2 | FAO | South America (Andean Mountains) ^d |
| Sweet potatoes | 2 | FAO | South America (Peru, Equador) ^e |
| Millet and sorghum | 2 | FAO | China, Abyssinia; Abyssinia ^f |
| Beans | 2 | Est. | Central America ^g |
| Banana/plantain | 2 | Est. | SE Asia, Western Pacific ^h |
| Cassava | 2 | Est. | South America (Brazil, Peru) ⁱ |
| Pigeon pea | 2 | Est. | India ^f |
| Lentils | 2 | Est. | Near East ^j |
| Cowpea | 2 | Est. | India, Abyssinia ^f |
| Yam | 2 | Est. | West Africa, Asia ^e |
| Proso millet | 2 | Est. | Eastern or Central Asia ^j |
| Peanut (groundnut) | 2 | Est. | South America (Brazil) ^g |

^aGibson, L. and Benson, G. Origin, History and Uses of Oat (*Avena sativa*) and Wheat (*Triticum aestivum*). Department of Agronomy, Iowa State University. January 2002.

^bZhao, Z. New Evidence on Rice Origin. *Agricultural Archaeology* 1998(1):394.

^cAdvanced Maize. Monsanto in India. <http://www.monsantoindia.com/asp/facts/maize/advmaizeorigin.asp> (6/12/03).

^dPotato Info and FAQ. Sun Spiced. <http://www.sunspiced.com/phistory.html> (6/11/03).

^eSchultheis, J. and Wilson, L.G. What is the Difference Between a Sweetpotato and a Yam? North Carolina State University. Revised 1993. www.ces.ncsu.edu/depts/hort/hil/hil-23-a.html (6/11/03).

^fWorld Centers of Origin of Cultivated Plants. <http://ucdnema.ucdavis.edu/imagemap/nemmap/ENT135/expl.htm> (6/11/03).

^gSeeds of Change Garden. <http://www.mnh.si.edu/garden/history/> (6/11/03).

^hARC–Institute for Tropical and Subtropical Crops (ARC-ITSC). <http://www.arc.agric.za/institutes/itsc/main/banana/origin.htm> (6/12/03).

ⁱOlsen, K.M. and Schaal, B.A. Evidence on the origin of cassava. *Proc. Nat. Acad. Sci. U. S. A.* 96 (1999): 5586–5591.

^jMuehlbauer, F.J., Tullu, A. *Lens culinaris*. *Medik: NewCROP FactSHEET*. Center for New Crops and Plant Products, Purdue University. 1997. (From Ref.^[6].)

Table 2 Major livestock groups that provide approximately 90% of the U.S. and World's DES from livestock

| Livestock | %DES | Source | Origin |
|-----------------------|------|--------|-----------------------------------|
| Cattle | 30 | Est. | Near East ^a |
| Sheep | 20 | | Asia, Europe ^a |
| Poultry (chickens) | 15 | | Southeast Asia ^b |
| Hogs | 15 | | Eurasia, Middle East ^a |
| Goats | 10 | | Asia ^a |
| Buffalo | 10 | | Asia, Africa ^a |

^aBreeds of Livestock. Department of Animal Science. Oklahoma State University. <http://www.ansi.okstate.edu/breeds> (6/20/03).

^bCrawford, R.D. Origin and history of poultry species. In *Poultry Breeding and Genetics*; Crawford, R.D., Ed.; Elsevier: Amsterdam, 1990; 1–41.

RISKS OF LOW DIVERSITY IN CROPS AND LIVESTOCK

The low diversity of world crops (0.006% of the plant species) and world livestock (0.1% of mammal species) presents the benefit of increased efficiency, but also risks such as increased vulnerability to pests and diseases. To protect crops (particularly in monoculture settings) and to pursue high yields, industrial farms often increase the amounts of pesticides, herbicides, fungicides, and fertilizers; soil erosion and depletion often result. There are also public and farm-worker health risks associated with chemical-intensive farming. The estimated economic impact of human pesticide poisoning and other pesticide related illness in the United States each year is over \$1 billion.^[9]

In the United States the annual cost in losses and control efforts due to pests and disease is approximately \$100 billion for crops^[10] and \$20 billion for livestock. Also, the world loses 40% of food production annually despite about 3 billion kg of pesticide applied. Biodiversity offers some natural protection from pests and disease pathogens and also protects pollinators that are essential for about one third of U.S. and world crops.

The current threat of bioterrorism has brought a call from the American Association of Veterinary Laboratory Diagnosticians to establish an Animal Disease Diagnostic Network that would link local, state, and federal (USDA) resources in a communication system to enhance quick response to natural and or intentional contamination of livestock. The proposed startup cost of this network is estimated to be \$85 million with yearly additional costs of \$22 million.^[11]

United States port inspections find 13,000 exotic plant diseases a year while checking only 2% of incoming freight. Both crops and livestock are vulnerable, especially because in each case we depend upon a narrow band of species.

While genetic engineering holds promise for protecting crops and livestock from diseases, it also carries risks. Wheat varieties, for example, may be protected from fungal diseases for several years before they are threatened again by a mutated version of the same pathogen. However, hybrids can threaten other crops, even original, wild strains. Researchers in Mexico have found that wild maize has been contaminated by genetically modified crops.^[12] The research is controversial, but the threat is evident. To protect plant varieties 6 million samples are preserved in 1000 gene banks worldwide.^[10]

CONCLUSION

Fifteen non-indigenous crops and six non-indigenous livestock species provide approximately 90% of the food supply in the United States and the world. The introduction and domestication of these species have had benefits and led to efficiency for producers. However, as modern agriculture has chosen a relatively small number of plants and animals to domesticate, these chosen species have become the dominant plant and animal species on earth following the rise of human domination. This agricultural phenomenon has impacted the global environment, replacing natural ecosystems, rich in biodiversity, with vast areas of intentionally simplified and disturbed agricultural regions.^[13] Monoculture regions have displaced ecosystems that once held thousands of plant, animal, and microbe species.

The lack of diversity of domesticated non-indigenous species results in a vulnerability to arthropod pests, weeds, and diseases. Increased use of chemicals and nutrients impact the environment and soil negatively. As the human population of 6.2 billion will double in 50 years,^[14] for a more sustainable agriculture we need to creatively diversify our agricultural base and minimize environmental harm.

REFERENCES

1. USBC. United States Bureau of the Census. *Statistical Abstract of the United States*; U.S. Government Printing Office: Washington, DC, 2002.
2. WHO. World Health Organization. *Micronutrient Malnutrition—Half the World's Population Affected*; Press Release WHO No. 78; World Health Organization: Geneva, 1996; 1–4.
3. WHO. World Health Organization. *Malnutrition Worldwide*; 2000. http://www.who.int/mipfiles/2299/MIP_01_APR_SDE_3.en.pdf (accessed July 2000).
4. Sommer, A.; West, K.P. *Vitamin A Deficiency: Health, Survival and Vision*; Oxford University Press: New York, 1996.

5. Tomashek, K.M.; Woodruff, B.A.; Gotway, C.A.; Bloand, P.; Mbaruku, G. Randomized intervention study comparing several regimens for the treatment of moderate anemia refugee children in Kigoma Region, Tanzania. *Am. J. Trop. Med. Hyg.* **2001**, *64* (3/4), 164–171.
6. FAO. *FAO Q. Bull. Stat.*; Food and Agriculture Organization of the United Nations: Rome, Italy, 1961–1999.
7. PRB. Population Reference Bureau. *World Population Data Sheet*; Population Reference Bureau: Washington, DC, 2002.
8. Pimentel, D.; Doughty, R.; Carothers, C.; Lamberson, S.; Bora, N.; Lee, K. Energy inputs in crop production in developing and developed countries. In *Food Security and Environmental Quality in the Developing World*; Lal, R., Hansen, D., Uphoff, N., Slack, S., Eds.; CRC Press: Boca Raton, 2002; 129–151.
9. Pimentel, D. Environmental and economic costs of the application of pesticides primarily in the United States. *Environment, Development and Sustainability* **2005**, *7* (2), 229–252.
10. Margolis, M. Crisis in the cupboard. *Newsweek* **2003**, *CXLI* (23), 36–40.
11. McElwain, T.F. *Establishing an Animal Disease Diagnostic Network*; Agricultural Outlook Forum: Arlington, VA, February 20, 2003. American Association of Veterinary Laboratory Diagnosticians.
12. Quist, D.; Chapela, I. Transgenic DNA introgressed into traditional maize landraces in Oaxaca, Mexico. *Nature* **2001**, *414*, 541–543. (29 Nov).
13. Tilman, D. Global environmental impacts of agricultural expansion: the need for sustainable and efficient practices. In *Plants and Population: Is there Time?*; Proc. Natl. Acad. Sci. U.S.A., National Academy Press: Washington, DC, 1999; Vol. 96, 5995–6000. National Academy of Sciences Colloquium, Beckman Center, Irvine, CA, December 5–6, 1998 (<http://www.lsc.psu.edu/NAS/Speakers/Tilman%20manuscript.html>).
14. Pimentel, D.; Pimentel, M. World population, food, natural resources, and survival. *World Futures* **2003**, *59* (3–4), 145–167.

Non-Indigenous Species: Pests

J. Howard Frank

Department of Entomology and Nematology, University of Florida,
Gainesville, Florida, U.S.A.

INTRODUCTION

About 50,000 adventive (= non-indigenous) species are now present in the U.S.A.^[1] These include the species that are the basis for 98% of the country's agriculture as crops and livestock. They include the species that form all of the pet animal and most of the ornamental plant businesses. They include large numbers of innocuous species. They also include pests (including weeds and pathogens) that cause damage and losses totaling about \$137 billion annually.^[1] Some of these pests were introduced (deliberately) by bad judgment on the part of the importers and insufficient governmental oversight, but many are immigrants which arrived by natural range extension, or as hitchhikers, stowaways, or contaminants aboard vehicles and insufficient governmental detection at ports of entry. This account is of the damage that adventive pests cause in the U.S.A.; comparable worldwide data are not available.

BACKGROUND

For the last two decades, authors of a clutter of publications fished for a word. What they wanted was a word to mean "of foreign origin but now present here." At first they used "non-indigenous," which is fairly accurate, but klutzy. More recently they use "exotic," which is short but inaccurate because it does not imply a presence "here." **Adventive**, a word dating from 1605,^[2-5] means "arrived here from somewhere else," and is the right word. Another word used inappropriately is "introduced," which implies human action, whereas many adventive species arrive as immigrants, without human action or at least without deliberate human action. If you have not before heard the expression **immigrant** applied to animals^[6] and plants, then you have not paid attention to the relevant literature of the last 22 years. Nor have you considered how it is that the Hawaiian Islands, which emerged by volcanic action from the seabed beginning some 5 million years ago, were populated by plants and animals before the first humans (Polynesians) arrived

there. **Adventive** is the complement of **native**, and is used here instead of non-indigenous. **Introduced** is here reserved for those organisms that were introduced deliberately.^[7]

Great problems were generated by under-regulated commercial importations, especially of plants and mammals, and some of these spilled over to cause environmental problems. This happened in part because some of the imported organisms were capable of becoming pests if they escaped, and they did escape. It happened in part because some of the imported organisms (and other materials) arrived contaminated by diseases, parasites, pest insects, and weed seeds. Commerce generates wealth, but it seeks public aid when it faces problems (as in the examples above) and leaves resultant environmental problems to be solved at public expense or not at all. Human error caused much of the \$137 billion annual losses, but not all. Some plant pathogens and insects are borne long distances on high-altitude winds, and various organisms disperse shorter distances by rafting on sea drift and by flight. Walking is an entry method, across the Mexican and Canadian borders.

Some adventive species are beneficial. Most are innocuous. Even among those that are labeled pests, the effect of some is equivocal. Thus, red imported fire ant (*Solenopsis invicta*) is harmful in several ways, but is an important predator of various pest insects. Even the much-maligned zebra mussel and kudzu are in some ways beneficial.^[8]

A 1993 report^[9] showed the United States Department of Agriculture's Animal and Plant Health Inspection Service (USDA-APHIS) spent \$80 million, and the United States Fish and Wildlife Service (FWS) spent \$3 million, in inspections at U.S. ports of entry, without data for inspection by the United States Public Health Service. The level of interdiction of adventive species achieved is trivial (fewer than 2% of shipments are inspected). A tenfold increase in inspections might begin to stem the tide of adventive species and would make economic sense, but is unlikely to be voted funds by the U.S. Congress—it would raise government spending, and would raise the wrath of travelers and commerce alike if inspection caused delays in transit of passengers and commercial shipments.

NATURAL ENVIRONMENTS (MARINE, FRESHWATER, TERRESTRIAL)

All of the following cost estimates (in parentheses) are from one source.^[1] In marine environments, various shipworm species (\$205 million) arrived as contaminants, but the European green crab (\$44 million) was introduced as a food source. In freshwater environments, Asian clam (\$1 billion) and zebra mussel (\$100 million) arrived as contaminants, although the former was initially introduced to Canada by Asian laborers and may have been helped to spread by the bait and aquarium industries.^[10] Numerous species of “sport” fishes (\$1 billion) were introduced, even by the U.S. Fish and Wildlife Service. Several species of aquatic and semiaquatic weeds (\$160 million) were introduced by the aquarium trade or as ornamental plants. In terrestrial environments, mammals (\$18 billion) were probably all introduced. The total damage cited is over \$20 billion. However, that total is by no means all: The damage and cost of control of non-terrestrial plants that cause environmental damage are included.^[1] Insects causing environmental damage are not included. The total is an underestimate. Some funds have been expended to prevent brown tree snake from hitchhiking from Guam to the U.S.A.

FORESTS

Adventive plant pathogens and insects are each estimated to cause \$2.1 billion annually in losses to U.S. forests.^[1] The pathogens include chestnut blight fungus and Dutch elm disease. Gypsy moth, European elm bark beetle (which spreads Dutch elm disease), European pine shoot moth, balsam woolly adelgid, and hemlock woolly adelgid are among the pest insects.

HUMAN HEALTH

Pathogens of foreign origin such as AIDS and influenza result in \$6.5 billion in losses and control costs annually.^[1] Perhaps the cost of controlling adventive mosquitoes such as *Aedes aegypti* and *A. albopictus* should be added to the total, because these have the potential to transmit important diseases, among which dengue is now epidemic in countries south of the U.S.A. The mosquito *Culex quinquefasciatus* is an adventive vector of (mainly) native viral encephalitides which cause illness and deaths in humans and horses during sporadic epidemics. The losses caused by and costs of control (\$1 billion) of red imported fire ant^[1] are perhaps more appropriately placed here than in any other category, but it also harms livestock and wildlife.

LIVESTOCK HEALTH

Adventive pathogens and parasites, including blood-sucking insects and arachnids, cause \$9 billion annually in damage and losses to livestock production.^[1] Four fly species—house fly, horn fly, stable fly, and face fly, all of European or North African origin—contribute to those losses. Even domestic (including feral) dogs, which are of Old World origin, contribute \$10 million annually in livestock losses.^[1]

PET ANIMALS

Protection of dogs and cats from cat fleas (*Ctenocephalides felis*), a species of Old World origin, now brings pharmaceutical companies about \$0.5 billion in sales in the U.S.A.

RANGELAND, PASTURES, TURF, AND GARDENS

Adventive weeds, plant pathogens, and insects, contribute respectively \$7.5, \$2.0, and \$1.5 billion in losses to cultivated grasses and ornamental plants.^[1] Among the insect offenders that attack grasses are South American *Scapteriscus* mole crickets, Japanese beetle, and European crane fly.

FIELD AND GLASSHOUSE CROPS, INCLUDING FRUIT TREES

Huge losses and control costs are experienced annually by farmers due to adventive weeds (\$26.4 billion), plant pathogens (\$21.5 billion), and insects (\$14.4 billion); the control costs alone amount to \$4 billion of the above total, and European starlings (\$0.8 billion) contribute additional losses.^[1] Some U.S. funds have been expended for biological control of pink hibiscus mealybug (*Maconellicoccus hirsutus*) in the Caribbean, in anticipation of its eventual arrival in the U.S.A.; this is an unusual case in which APHIS personnel have been proactive^[11]—their few inspectors at U.S. ports of entry are normally the first line of defense against arrival of new pests.

HONEY BEES

The adventive pest problems faced by the honey bee industry include Varroa mite, honey bee tracheal mite, beelice, small hive beetle, and European foulbrood; there seems to be no available figure for losses and costs of control to the industry. African honey bees

escaped from confinement in Brazil and established feral populations which extended their range, and eventually crossed the Mexican/U.S. border to colonize the U.S. southwest. Detection and eradication are in progress, but there seem to be no published estimates of costs, nor of losses caused by stings from these bees.

STORED PRODUCTS

Losses and damages caused by adventive rats (\$19 billion)^[1] are here attributed to stored products. Major losses to stored products are caused by insects and mites, many of which are adventive, but estimates of those losses and of control costs seem unavailable.

DWELLINGS AND OTHER STRUCTURES, AND VEHICLES

Formosan termite (\$1 billion) and pigeons (\$1.1 billion) are two of the most important adventive pests.^[1] Expenditures on over-the-counter control materials used against the German cockroach amount to about \$800 million.^[12] Damages, and costs of professional exterminators controlling that cockroach, other cockroaches, and various other pests, have not been estimated.

CONCLUSION

Every imported shipment of plants and animals (and other materials) brings with it a risk of the arrival of additional pest species. Every arriving international traveller and migrant bird raises the risk of arrival of additional diseases. The number of adventive pest

species in the U.S.A. will continue to grow. The one thing that could stem the tide is greatly increased inspection and interdiction at ports of entry.

REFERENCES

1. Pimentel, D.; Lach, L.; Zuniga, R.; Morrison, D. Environmental and economic costs of non-indigenous species in the United States. *BioScience* **2000**, *50*, 53–65.
2. Bacon, F. The two books of francis bacon, of the profience and advancement of learning, divine and humane. Henry Tomes: London, 1605.
3. Pemberton, C.E. Highlights on the history of entomology in Hawaii 1778–1963. *Pacific Inst.* **1964**, *6*, 689–729.
4. Frank, J.H.; McCoy, E.D. Endemics and epidemics of shibboleths and other things causing chaos. *Fla. Entomol.* **1990**, *73*, 1–9.
5. Frank, J.H.; McCoy, E.D. Precinctive insect species in Florida. *Fla. Entomol.* **1995**, *78*, 21–35.
6. Sailer, R.I. Our immigrant insect fauna. *Bull. Entomol. Soc. Am.* **1978**, *24*, 3–11.
7. Zimmerman, E.C. *Insects of Hawaii*. Univ. Hawaii Press: Honolulu, 1948; Vol. 1.
8. Sagoff, M. Why exotic species are not as bad as we fear. *Chron. Higher Educ.* **2000**, *46* (42), B7.
9. US Congress, Office of Technology Assessment *Harmful Non-Indigenous Species in the United States*; Govt. Ptg. Office: Washington, DC, 1993; OTA-F-565.
10. Warren, G.L. Nonindigenous freshwater invertebrates. In *Strangers in Paradise*; Simberloff, D., Schmitz, D.C., Brown, T.C., Eds.; Island Press: Washington, DC, 1997; 101–108.
11. Hoy, M.A.; Hamon, A.B.; Nguyen, R. *Maconellicoccus hirsutus* (Green) (Insecta: Homoptera: Pseudococcidae), <http://www.ifas.ufl.edu/~insect/ORN/mealybug/mealybug.htm> (accessed July 2000).
12. Koehler, P.G. Entomology and Nematology Department, University of Florida, personal communication, June, 2000.

No-Till and Pest Problems

Punnee Soonthornpoch

Department of Natural Sciences, Blinn College, Bryan, Texas, U.S.A.

INTRODUCTION

Conventional tillage operations are considered a major step in crop production. The practice involves change of soil condition for seedbed preparation and for weed control. However, conventional tillage has been documented to cause soil erosion, contamination of surface and groundwater, and organic matter loss. Annual tillage can increase soil erosion from 10 to 1000 times. For the past decades, interest has turned to the practice of conservation tillage in which at least 30% of crop residues are left on the soil surface. No-till is a type of conservation tillage in which new crops are planted on untilled soil. The presence of total previous crop residues along with the new crops may create problems with pest control. Previous crop residues may serve as overwintering reservoir for several plant pathogenic fungi, as safe harboring for the next generation of insects inhabiting in the soil, and as suitable environment for the germination of weed seeds left near the soil surface.

NO-TILL SYSTEM

During the past decades, the rising cost of fuel, labor, and machinery, and environmental concerns have caused farmers to consider alternative agricultural methods. The introduction of plant growth regulators and their development into selective herbicides has generated interest in conservation tillage as an alternative method to prevent soil erosion without jeopardizing the weed control normally accomplished by overturning of the soil. Conservation tillage system aids in reducing soil erosion by leaving at least 30% of the soil surface covered by previous crop residues.

Conservation tillage systems include strip-till, mulch-till, ridge-till, reduced-till, and no-till. No-till was defined by the Conservation Technology Information Center as: "The soil is left undisturbed prior to planting. Planting is completed in a narrow seedbed or slot created by a planter or drill. Weed control is accomplished primarily with herbicides. Cultivation may be used for emergency weed control."

The benefits of conservation tillage have been well established: conserving soil moisture, protecting the topsoil, maintaining soil quality, and saving fuel and labor cost. The systems have been established in many

areas of the world, especially in areas where soil moisture and erosion influence crop production. The practice of no-till is adopted readily by many farmers who are faced with delayed planting because of cold temperature or high moisture. No-till system offers the opportunity to plant without waiting for sufficient drying time for the soil normally required for conventional tillage. No-till tropical maize grown during late spring and summer in the southern United States has the advantages of summer rain and predictable early fall drought in time for maturity and harvest.

EFFECTS OF NO-TILL ON WEED MANAGEMENT

Several factors influence the efficiency of weed management programs in the no-till system: weed species, soil type, herbicide selection, and environmental condition. The effectiveness of herbicides in no-till is reduced in years following the failure of weed seed control. The number of weed seeds left in the soil generally determines potential population of weed in a field. Intensive conventional tillage program distributes and buries weed seeds along the plow depth. In the no-till system, weed seeds are left undisturbed on or very near soil surface. Soil moisture protected by the presence of previous crop residues creates perfect environment condition for seed germination of several of these weed species. Others, such as foxtail, prefer to germinate in deep soil profile created by conventional tillage and therefore do not thrive in no-till. Several weed species are not affected by type of tillage. Among these are lambsquarter, smooth pigweed, and fall panicum.

Failure of herbicides to control the cool-season grass when applied at late winter planting forces researchers to search for other possible method. Late winter no-till establishment of ladino clover proved to be an alternative to their fall planting. Ladino clover planted into no-till tall fescue residue thrived successfully.

EFFECTS OF NO-TILL ON INSECT PROBLEMS

Nearly 90% of terrestrial insect species pass part of their lives in soil or on the soil surface. Among these

are insects important to grain crops such as corn earworm (*Heliocoverpa zea*), European corn borer (*Ostrinia nubilalis*), black cutworm (*Agrotis ipsilon*), wheat stem sawfly (*Cephus cinctus*), and Hessian fly (*Mayetiola destructor*). Conventional-tillage practice may kill several soil-dwelling insects that are present in the upper soil layer. A no-till system where the residues are left undisturbed creates a cooler, moister soil environment that may slow the early growth of the grain crop and become susceptible to feeding by early-season soil insects.

European corn borers overwintering as diapause inside corn stub can survive a long period of winter until adults emerge. In this case, the control measure recommended is to destroy the crop residues. In conventional-tillage practice, plowing down stubbles and residues has been reported to reduce the number of overwintering of *O. nubilalis* larvae nearly 90%. Second deep plowing prevents the emerging moths from reaching the soil surface. The unincorporated residues in the no-till system provide favorable site for survival and colonization for these soil insects. Hence for the area that has history of infestation of either corn earworm or European corn borer, the no-till system of tillage is usually not recommended. In the Midwestern corn belt area, where loss of corn because of soil insects is generally not significant enough for use of insecticides at planting in the conventional tillage, the suppression of soil insects, such as European corn borers and wireworms (*Monocrepidius vespertinus* Fab.), with the application of insecticides at planting time is necessary for the no-till system. The second application within a day of egg hatching, coupled with good weed management program, may help to minimize the damage. Early harvest from the infested no-tilled fields will help to minimize loss from stalk breakage. In this case, mowing after harvest is recommended. The use of corn stalks for silage in some areas results in the removal and the destruction of larvae from the infested field. In the no-till wheat crop, the Hessian fly populations are carried over in wheat stubble, especially in the area where volunteer wheat is not controlled. The presence of wheat residues may deter the invasion of some airborne aphids.

Corn is the most widely grown in the no-till system. Rotation of corn with soybeans has great impact on variety of insects, notably corn rootworms, wireworm, and white grubs. The use of crop rotation generally results in the reduction of population of these soil insects because of the change in their sustainable host environment. In the Corn Belt area, rotation results in the reduction in pesticide usage. However, in a phenomenon that has not yet been understood, researchers have discovered that populations of corn rootworms in Illinois and Indiana have changed their behavior and now lay their eggs in the soybean fields.

In this case, rotation has very little effect. Fortunately, this problem only occurred in these two states. In cotton, cutworms are normally not a problem in a conventional-till system. Rotation of cotton with legume crops in the no-till system would be fatal because cutworm larvae will attack cotton leaves and destroy whole seedlings as they mature.

Researches have indicated that the reduced tillage system favors the predatory insects that feed on eggs of soil insect pests of corn. *Coleomegilla maculate* DeGeer and several species of lady beetles feed on eggs of *H. zea*. The fact that the no-till system has positive effects on the population of soil insects may indicate that it plays a role in influencing the population of the predators. However, the correlation between predator number and predation is not always stable. Other factors, such as prey and predator density, characteristics of prey and predators, and characteristics of the environment, may be accountable in this relationship.

EFFECTS OF NO-TILL ON DISEASE PROBLEMS

In the no-till system, surface residue from the previous crops alters the soil temperature. As the soil temperature rises slowly in comparison to the soil in conventional tillage, a seedling develops slowly and may be conducive to damping-off and root diseases induced by soilborne pathogens that favor low temperature, such as several species of *Pythium* spp. Several foliar pathogens are known to survive in crop residues, which then produce conidia as the primary source of inoculum. Several foliar diseases of maize, such as northern and southern corn leaf blight (incited by *Helminthosporium turcicum* and *Helminthosporium maydis*), yellow corn leaf blight (incited by *Phyllosticta maydis*), and gray leaf spot (incited by *Cercospora zeae-maydis*), are more severe in minimal tillage than when maize debris is buried by plowing.

Several seedling diseases are not influenced by the system of tillage practice. Inoculum density of the soilborne fungi *Rhizoctonia solani* AG-4, incitant of seedling disease of vegetable, is not influenced by the tillage method. The pathogen is more influenced by the presence of susceptible host. The frequency of isolation of *Fusarium* spp. was reported to be lower in the no-till system than in the conventional system. However, root rot of corn caused by *Pythium ultimum*, an organism that is ubiquitous in agricultural soils, is a serious disease in the cold-wet soil of no-till systems.

In the cold-wet climate of the northwestern United States, the incidence of take-all disease of wheat, incited by *Gaeumannomyces graminis* var. *tritici*, is significantly decreased in the minimal tillage. The lack of late season moisture stress was also contributed to

the lower incidence of stalk rot of maize in a no-till system than when conventional tillage was used. Tillage did not have any significant effect on the colonization of maize seedling roots by several species of soilborne fungi. However, the population of *Fusarium* spp. in the silty loam soil in Mississippi was higher in the tilled plots than in the no-tilled plots.

Reduced tillage has been shown to increase the incidence of southern stem canker of soybean incited by *Diaporthe phaseolorum* var. *caulivora*. The lower disease incidence in the conventional tillage is contributed to the lower inoculum level when the residues are buried in the tillage treatment.

FUTURE CONCERNS

While the usage of no-till and minimum tillage helps save costs of labor and fuel, the cost of herbicide is higher than that of convention tillage. From the overview angle, nevertheless, the farmers still see good profit. The unseen profit lies in the conservation of soil. The problems of no-till concerning the management of weeds rest on the cost of herbicides because weed control depends entirely on chemicals. To reduce tillage but increase the quantity of herbicide will defeat the purpose of conservation. More research is needed in the future to direct attention to the use of rotation crops that will be able to compete with the presence of weeds. Planting crops into previous crop stubble in the no-till system should have minimal direct effect on insect population. However, the low soil temperature environment may create a delay in maturity and often increases the potential damage from insect pests because of the extended season.

Conservation tillage causes an increase in insect populations such as cotton bollworm and tobacco budworm. Future concern should be placed on using the resistant hybrids. It has been predicted that the use of transgenic corn hybrids with insect resistance will be more common in the future. Disease problems with no-till can be avoided by choosing the planting time that will give seedlings the optimum development. The use of fungicide seed treatment and resistant varieties is highly recommended.

BIBLIOGRAPHY

- 50 Ways Farmers Can Protect Their Groundwater; University Extension, University of Missouri—Columbia, 1999. Agricultural Publication MX397. http://www.thisland.uiuc.edu/50ways/50ways_10.html.
- Anonymous U.S. farmers choose environmentally beneficial growing system. *Conserv. Impact* **1995**, *13*, 4.
- Boosalis, M.G.; Douppnik, B.; Odvody, G.N. Conservation tillage in relation to plant disease. In *CRC Handbook of Pest Management in Agriculture*; Pimentel, D., Ed.; CRC Press: Boca Raton, FL, 1981; 1, 445–474.
- Buhler, D.D.; Mester, T.C. Effect of tillage systems on the emergence depth of giant (*Setaria faberi*) and green foxtail (*Setaria viridis*). *Weed Sci.* **1991**, *39*, 200–203.
- Monks, C.D., Patterson, M.G., Eds.; *Conservation Tillage: Cotton Production Guide*; Circular ANR-952. Alabama Cooperative Extension System, Alabama A&M and Auburn University.
- Eisley, B.; Hammond, R. *Insect Pests of Field Crops*; Ohio State University: Columbus, OH, Bulletin #545.
- Lukemann, W.H. Insect control in corn—Practices and projects. In *Pest Control Strategies*; Smith, H., Pimentel, D., Eds.; Academic Press: New York, 1978; 137–155.
- Phillips, S.H. Introduction. In *No-Tillage Agriculture*; Phillips, R.E., Phillips, S.H., Eds.; Van Nostrand Reinhold Co.: New York, 1984; 1–11.
- Sailer, R.I. Extent of biological and cultural control of insect pest of crops. In *CRC Handbook of Pest Management in Agriculture*, 2nd Ed.; Pimentel, D., Ed.; CRC Press: Boca Raton, FL, 1991; Vol. 1, 3–12.
- Soonthornpoc, P. Partial characterization of soilborne fungi associated with roots of maize symptomatic for seedling disease in Mississippi. Ph.D. diss., Miss. State Univ., Miss State, MS, 1998.
- Soonthornpoc, P. The colonization of maize seedling roots and rhizosphere by *Fusarium* spp. in Mississippi in two soil types under conventional tillage and no-tillage systems. *Phytoprotection* **2000**, *81*, 97–106.
- Soonthornpoc, P.; Tevathan, L.E.; Gonzalez, M.S.; Tomaso-Peterson, M. Fungal occurrence, disease incidence and severity, and yield of maize symptomatic for seedling disease in Mississippi. *Mycopathologia* **2000**, *150*, 39–46.
- Sumner, D.R.; Douppnik, B.; Boosalis, M.G. Effects of reduced tillage and multiple cropping on plant disease. *Annu. Rev. Phytopathol.* **1981**, *19*, 167–187.
- TAMU. *1999 Projected Costs and Return per Acre*; 1999. <http://www.agecoext.tamu.edu/budgets.list.htm> (October 18, 1999).
- Triplett, G.B.; Dabney, S.M.; Sourlock, S.R.; Reinschmiedt, L.L. Management system for soil conservation and sustainable crop productivity. In *Proceeding of the Conference on Management of Landscapes Disturbed by Channel Incision*; Wang, S.S.Y., Wand, E.G., Langendoen, E.J., Shields, F.D., Jr., Eds.; University of Mississippi: Oxford, MS, 1997; 1087–1092.
- Whitcomb, W.H. The use of predators in insect control. In *Handbook of Pest Control Management in Agriculture*; Pimentel, D., Ed.; CRC Press: Boca Raton, FL, 1981; Vol. 2, 105–203.
- Whitcomb, W.H.; Bell, K.O. Predation. In *The Bollworm-Tobacco Budworm Problem in Arkansas and Louisiana*; Lincoln, C., et al., Eds.; Bulletin, 1967; Vol. 720, 34. Agric. Exp. Stn. Univ. Arkansas, Fayetteville; La. Agric. Exp. Stn.
- Wright, R.J. *Corn Production: A Guide to Profitable and Environmentally Sound Management—Insect Management*; University of Nebraska.

Nozzle Types

Alvin R. Womac

Department of Agricultural and Biosystems Engineering, University of Tennessee, Knoxville, Tennessee, U.S.A.

INTRODUCTION

Spray nozzles atomize aqueous- or oil-based mixtures of various agrochemicals, herbicides, insecticides, fungicides, and nutrients to coat the targeted weed, insect pest, crop foliage, or soil furrow. Spray application trends, including increased droplet size spectra to reduce off-target spray drift, influence the development and selection of nozzle types.

DROPLET SIZE SPECTRA

A spray nozzle produces a spectrum of droplet sizes at any instant while spraying. Look-up charts^[1] and research articles^[2–5] provide specific droplet size spectra data for various nozzle types, sizes, operating pressures, and spray mixtures. A new ASAE standard classifies complex droplet spectra into user-friendly categories ranging from very fine to extremely coarse.^[6–8] Droplet size spectra influence spray drift,^[9–10] spray deposition efficiency,^[11] and product biological efficacy.^[12]

NOZZLE TYPES

ASAE Standard S327.2^[13] lists at least 34 categories of atomizing devices based on energy for atomization and geometric configuration of orifices, chambers, and deflectors.

Single-Elliptical-Orifice Nozzles

Single-elliptical-orifice nozzles producing a fan pattern are widely used because of proven simplicity and low cost. Fan patterns are typically sustained at pressures as low as 100 kPa through use of extended pressure range designs. Volume median diameters ($D_{v0.5}$) and the spray volume in droplets less than 100 μm typically range from about 130–350 μm and 3–30%, respectively.^[1]

Deflector/Flooding Nozzles

Single orifice, deflector outlet flooding nozzles produce a wide fan pattern at low boom heights and are rugged

and low cost. Flooding nozzles operate at very low pressures (69 kPa) and often create coarse sprays ($D_{v0.5} \sim 300+ \mu\text{m}$).^[1] They are typically used for soil-incorporated applications that have a low boom height.

Pre-Orifice Nozzles

Pre-orifice nozzles use a metering orifice upstream from the exit orifice. This arrangement tends to increase the emitted droplet size spectra by reducing the effective exit pressure, and by merging droplets in a chamber between the orifices. Exit orifices are typically an elliptic orifice, or circular with a flooding deflector. Pre-orifice nozzles moderately increase droplet sizes by approximately 15% compared to the exit nozzle alone.^[1] However, some pre-orifice nozzles develop $D_{v0.5}$ values greater than 1000 μm .^[14] Pre-orifice nozzles require greater operating pressure than single orifice nozzles. Multiple pre-orifices in series, or in parallel, are used. Contaminants are more prone to plug pre-orifice nozzles as compared to single orifice nozzles.

Venturi/Air-Induction Nozzles

Venturi nozzles are similar to the concept of pre-orifice nozzles, with the addition of an air inlet into a negative pressure chamber located between the metering orifice(s) and exit orifice. Venturi nozzles typically produce extremely coarse droplets ($D_{v0.5} \sim 600 \mu\text{m}$)^[1,5] that contain varying degrees of entrained air bubbles within emitted droplets. The amount of entrained air largely depends on the product being sprayed. Many unsubstantiated claims are made about the advantages of entrained air bubbles. Venturi nozzles typically require 200 kPa of pressure to operate without a collapse in the fan pattern. This should be taken into account when using pressure-based sprayer rate controllers.

Other Nozzle Types and Considerations

Other typical nozzle types include hollow and solid cone nozzles, disk-core cone nozzles, pneumatic or air assist nozzles (independent control of spray rate

and droplet size), and rotary atomizers, spinning disks, or cups (uniform, fine droplet size). Also, many variations in pattern eccentricity are available for directed, offset, under-leaf, and other specific applications for many of the nozzle types discussed. Another aspect of spray nozzles involves secondary atomization due to air shear, such as nozzles on orchard, air blast, and aircraft.

REFERENCES

1. Womac, A.R.; Goodwin, J.C.; Hart, W.E. *Tip Selection for Precision Application of Herbicides, A Look-up Table of Drop Sizes to Assist the Selection of Nozzles*; TAES Bulletin, The University of Tennessee Agricultural Experiment Station: Knoxville, Tennessee, 1997; 695 pp.
2. Reeves, K.C.; Womac, A.R. *Reduced-Drift Characteristics of Pre-Orifice, Flat-Spray RF Raindrop Nozzles*; American Society of Agricultural Engineers: St. Joseph, Michigan, 1992; Paper No. 921612.
3. Womac, A.R.; Hart, W.E.; Maynard, R.A., II. Drop spectra for pneumatic atomizers at low discharge rates. *Trans. ASAE* **1998**, *41* (4), 941–949.
4. Mueller, T.C.; Womac, A.R. Effect of formulation and nozzle type on droplet size with isopropylamine and trimesium salts of glyphosate. *Weed Technol.* **1997**, *11*, 639–643.
5. Etheridge, R.E.; Womac, A.R.; Mueller, T.C. Characterization of the spray droplet spectra and patterns of four venturi-type drift reduction nozzles. *Weed Technol.* **1999**, *13* (4), 765–770.
6. ASAE S572 Spray Nozzle Classification by Droplet Spectra. In *ASAE Standards*, 47th Ed.; 2000; 389–391.
7. Womac, A.R.; Maynard, R.A.; Kirk, I.W. Measurement variations in reference sprays for nozzle classification. *Trans. ASAE* **1999**, *42* (3), 609–616.
8. Womac, A.R. Quality control of standardized reference spray nozzles. *Trans. ASAE* **2000**, *43* (1), 47–56.
9. Amin, M.K.; Womac, A.R.; Bui, Q.D.; Mueller, T.C.; Mulrooney, J.E. Air sampling of aerosol and gaseous pesticides. *Trans. ASAE* **1999**, *42* (3), 593–600.
10. Bui, Q.D.; Womac, A.R.; Howard, K.D.; Mulrooney, J.E.; Amin, M.K. Evaluation of samplers for spray drift. *Trans. ASAE* **1998**, *41* (1), 37–41.
11. Womac, A.R.; Mulrooney, J.E.; Scott, W.P. Characteristics of air-assisted and drop-nozzle sprays in cotton. *Trans. ASAE* **1992**, *35* (5), 1369–1376.
12. Womac, A.R.; Mulrooney, J.E.; Scott, W.P.; Williford, J.R. Influence of oil droplet size on the toxicity of bifenthrin from cotton to tobacco budworm. *Pestic. Sci.* **1994**, *40*, 77–83.
13. ASAE S327.2 Terminology and Definitions for Agricultural Chemical Application. In *ASAE Standards*, 47th Ed.; 2000; 165–168.
14. Womac, A.R. *Performance Characteristics of New and Existing CP Floater/Tractor Spray Nozzles*; American Society of Agricultural Engineers: St. Joseph, Michigan, 2000; Paper No. 001131.

Obsolete Pesticides: Management

Alemayehu Wodageneh

Prevention and Disposal, Obsolete Pesticide Stocks, Food and Agricultural Organization of the United Nations (FAO), Rome, Italy

INTRODUCTION

The discovery of chemical pesticides in the early 1930s was considered a window of opportunity to eliminate pests and enhance modern agriculture. In the developed world, great profits would be made; for the less developed continents, hunger would become history, as mass production of food became a common place. However, the opportunity had a limited life-span and would instead have far-reaching and devastating environmental and human health consequences. Huge quantities of obsolete pesticide stockpiles have accumulated over several decades currently posing serious health hazards.

The 1992 UN Rio Conference on Environment and Development helped to bring about a much greater awareness of the issues surrounding sustainable development, environmental protection, and hazardous waste management. The twelve most dangerous chemicals, mostly pesticides that pose a threat to the planet, have since been classified as Persistent Organic Pollutants (POPs). POPs are also Long-Range Transportable Air Pollutants (LRTAPs). These chemicals escape into the environment and are borne by wind and ocean currents to be dispersed in soil and water over the entire globe.

No country is free from obsolete stockpiles, but the gravity of the situation is more serious in the developing countries despite less pesticides are used. There is widespread unawareness of the inherent danger of pesticides, no facilities to handle or destroy the waste, no expertise, and, above all, no financial support. Unfortunately, the problem is on the increase. Good sounding words that are frequently talked about, such as stewardship, safe use, and responsible use, are far from being effective. In fact, they have become conduits to distribute more pesticides. Unless urgent action is taken to minimize the use of pesticides and to get rid the accumulated waste, the consequences will be increasingly serious, more complex, and environmentally irreversible.

PESTICIDES, OBSOLETE PESTICIDES, AND PESTICIDES STOCKS

What Are Pesticides?

The *International Code of Conduct on the Distribution and Use of Pesticides*^[1] defines pesticides as:

Any substance or mixture of substances intended for preventing, destroying or controlling any pest, including vectors of human or animal disease, unwanted species of plants or animals causing harm during, or otherwise interfering with, the production, processing, storage, transport, or marketing of food, agricultural commodities, wood and wood products or animal foodstuffs, or which may be administered to animals for the control of insects, arachnids or other pests in or on their bodies. The term includes substances intended for use as a plant growth regulator, defoliant, desiccant, or agent for thinning fruit or preventing the premature fall of fruit, and substances applied to crops either before or after harvest to protect the commodity from deterioration during storage and transport.

When Are Pesticides Obsolete?

Obsolete pesticides are stocked pesticides that can no longer be used for their intended purpose or any other purposes and, therefore, require disposal.^[2] Common causes of this situation include the following:

- Use of the product may be prohibited or severely restricted for health or environmental reasons (e.g., it may be banned, its registration withdrawn; or its status affected by other policy decisions by the Ministry of Agriculture or other authorized ministries).
- The product may have deteriorated as a result of improper or prolonged storage and can no longer be used according to its label specifications and instructions for use, nor can it easily be reformulated to become usable.

- The product may not be suitable for its intended use and cannot be used for other purposes, nor can it easily be modified to become usable.

A product has deteriorated when:

- It has undergone chemical and/or physical changes that result in phytotoxic effects on the target crop, or an unacceptable hazard to human health or the environment.
- It has undergone an unacceptable loss of biological efficacy due to degradation of its active ingredient and/or other chemical or physical changes.
- Its physical properties have changed to such an extent that it can no longer be applied with standard or stipulated application equipment.

Obsolete pesticides are also referred to as “pesticide waste.” This term has a broader definition than simply obsolete pesticides, because it also includes waste generated during the production of pesticides.

What Constitutes Obsolete Stocks?

Obsolete pesticide stocks comprise the following four major categories:

1. Pesticides in the form of liquids, powders, granules, emulsions, gases, etc.
2. Empty and contaminated pesticides containers (millions of these are left at the farm gates each year, with little or no attention paid to their potential impact on the environment and human health unfortunately, most end up being used for domestic purposes such as water or food storage).
3. Contaminated soil either at storage site or in the open.
4. Buried pesticides either in engineered landfills or in shallow open or closed pits. As burial is a temporary solution, almost all buried pesticide stocks subsequently need to be excavated and disposed of in an environmentally safe way, but often at much higher cost.

GRAVITY OF THE ISSUE AND PROBLEMS

Environmental, Health, and Social Implications

Obsolete pesticides are global environmental tragedy. They are direct results of decades of misuse and mishandling of pesticides, and their accumulation can be attributed to a range of factors (Table 1). Few countries are unaffected by the harmful environmental

Table 1 Reasons for accumulation of obsolete pesticides

Other than direct misuse and abuse, experience has shown that several factors contribute to the accumulation of stocks, of which the following are salient:

- Donations in excess of requirements or uncoordinated donations by several donors at the same time.
- Aggressive sales or promotion of pesticides by the pesticides industry; a lack of accurate assessments of pesticide requirements.
- The dumping of pesticides as a pretext of donations.
- The banning of products while pesticides are still in store.
- Lower pest incidence than expected.
- Insufficient storage capacity, poor or substandard pesticide stores.
- Inadequate storage management or stock-taking.
- The absence of pesticide legislation or inability to implement existing legislation.
- Inappropriate government decisions to request or procure pesticides that are not required (this situation frequently occurs when technical or qualified staff have not been consulted).
- Improper labelling of imported, purchased, or otherwise received pesticides.
- A product or pesticide being inappropriate for its intended use.
- Fraudulent administrative practices.
- Civil war.
- Overstocking of products with a short shelf-life.
- A lack of product knowledge.
- Government policy on trade liberalization or subsidy.
- A change in agricultural crops.
- Stocked products being replaced by newer products.
- The unsuitable packaging of pesticides.
- The introduction of nonchemical crop protection methods.

legacy of obsolete pesticide stocks. The most serious problems are to be found in the developing world, where there is little or no awareness of the inherent danger of pesticides or pesticide waste; a lack of expertise and facilities for their destruction or disposal; and, above all, insufficient financial resources to address these problems. Leaking and corroding metal drums and other containers filled with obsolete and dangerous pesticides litter the rural landscapes of developing countries and populated areas around the world. These chemical residues have become time bombs in the agricultural world they were designed to help. They seriously affect not only a nation’s agriculture sector and environment but also the health of its people and, consequently, its development. People who are poor and sick—particularly when constantly

exposed to pesticides—lose their capacity to work or are less interested in development activities. They even lack the stamina and the energy required even for the simple work needed for survival.

Types of Obsolete Pesticides

Obsolete pesticides involve all kinds of pesticides; moreover, 9 of the 12 chemicals currently identified as POPs, namely, aldrin, chlordane, dieldrin, DDT, endrin, heptachlor, mirex, toxaphene, hexachlorobenzene, are pesticides. The other three—PCBs, dioxins, and furans—are industrial chemicals. These nine pesticides are used widely and are commonly mixed with other pesticides in the soil and in storage, exacerbating the negative impact on the environment. It is often impossible to separate the POPs from the other, less harmful, pesticides, and devising separate solutions for their respective disposal requirements is rarely feasible. POPs are universally toxic and resist degradation in the environment. They have low water solubility but are highly soluble in lipids. They bioaccumulate in fatty tissues, are semivolatile, and, therefore, highly mobile—they can be transported over vast areas by the wind or ocean currents.

International Efforts to Address the Issue of Obsolete Pesticide Disposal

International awareness of the severe environmental problems caused by obsolete stocks has gradually increased. Many countries have become seriously concerned about the negative impact on human health and have sought the advice and assistance of the Food and Agriculture Organization of the United Nations (FAO).

In mid-1994, with donor support from the Government of the Netherlands, FAO embarked on a program to address the issue. The first initiative involved taking an inventory of stocks to determine the quantity and type of pesticides involved. The initial regions for these activities were to be Africa and the Near East. As the donor support was limited, coverage had also to be limited at least in the initial phase. At the time of writing (July 2001), inventories have been completed in at least 46 countries in Africa and nine in the Near East. Their findings were indicative and, by no means, exhaustive, as additional obsolete stocks are constantly being discovered in each of the countries concerned. For this reason, the inventories will need to be constantly revised and updated until disposal operations have been completed.

With limited financial support from other sources, an inventory-taking exercise is gradually extended to several countries in the Far East and Latin American

countries. Although this work is still ongoing, initial survey results have made possible a conservative global estimate of stocks in developing countries. The current global estimate now stands at 500,000 t, but the total is likely to be higher if all types of stocks (i.e., pesticides, empty and contaminated containers, contaminated soil, and buried pesticides) are taken into consideration. Unfortunately, funds are not available to dispose of this estimated total which at an average disposal cost of \$3000 t⁻¹ will require at least \$1.5 billion.

Despite the many efforts made by FAO and a number of collaborative agencies, less than 3000 t have been disposed of so far (Table 2).

Methods of Disposal of Obsolete Stocks

The currently preferred method of disposal is to subject the pesticides to a high temperature in dedicated incinerators. This method is increasingly seen as inappropriate, especially by environmental non-governmental organizations, as incineration is likely to release dioxins into the atmosphere. Unfortunately, reliable alternatives that are also cheaper, better, and widely acceptable have yet to be developed. When suitable alternative methods are developed, the use of high-temperature incinerators will naturally phase out.

SOME USEFUL HINTS TO MINIMIZE THE ACCUMULATION OF STOCKS

The following measures, among others, can minimize the accumulation of obsolete stocks:

- Regulatory and administrative control on the use of pesticides.
- Control, restriction, and guidance of pesticides users both voluntary and statutory.
- Wider dissemination of information relating to pesticides and obsolete stocks.
- Public education through intensive use of the media.
- Reducing pesticide use through its replacement by alternative methods of pest control such as Integrated Pest Management (IPM) and, where possible, promoting organic agricultural methods that do not use pesticides.

THE FUTURE

Attempts to find solutions to the problems posed by obsolete stocks and their disposal on a country-by-country basis have so far been unsuccessful. Future

Table 2 Obsolete pesticide stocks disposed of in Africa and the Near East

| Country | Product | Quantity (t) | Agency involved |
|-------------------------|-------------------------|--------------|--|
| Niger ^a | Dieldrin | 60 | USAID and Germany |
| | | 70 | Germany |
| | | 65 | GCPF |
| Madagascar ^a | Dieldrin | 135 | Swiss and government |
| Uganda ^a | Dieldrin | 50 | FAO/UNCDF ^b |
| Mozambique ^a | DDT/Monocrotophos | 160 | Germany-GTZ |
| Tanzania (Zanzibar) | Various | 280 | The Netherlands-DGIS ^c /USAID |
| Seychelles | Various | 12 | FAO/the Netherlands-DGIS |
| Tanzania | Dinitro-O-Cresol (DNOC) | 57 | Germany |
| Yemen | Various | 262 | FAO/the Netherlands-DGIS/KfW ^d -Germany |
| Mauritania | Various | 200 | Germany-GTZ and (Shell contributed 37.5%) |
| Qatar | Various | 5 | Government |
| Zambia | Various | 360 | FAO/the Netherlands-DGIS/Germany-GTZ |
| Lebanon | Mainly fenitrothion | 10 | FAO 1999 |
| Iraq | Contaminated containers | 90,000 | FAO (different weights) 2001 |
| South Africa | Various | 603 | South African Government (1998/1999) |
| Swaziland | Various | 9 | South African Government (1998/1999) |
| Namibia | Mainly HCH | 202 | South African Government (1998/1999) |
| Gambia | Various | 14 | Industry and Overseas Development Corporation |
| Total | | 2,419 | |

^aDisposals undertaken in Uganda, Niger, Mozambique and Madagascar were limited either to dieldrin or few other types of stocks. There are still stocks in these countries that require disposal.

^bUnited Nations Country Development Fund.

^cMinistry of Foreign Affairs, Government of the Netherlands.

^dKreditanstalt für Wiederaufbau: (German Credit Bank in Frankfurt).

efforts must focus on developing a committed global effort to address these problems, with adequate financial resources to dispose of all stocks that have accumulated so far and to ensure the avoidance of further accumulations in the environment.

GUIDELINES RELATED TO OBSOLETE PESTICIDES

FAO has developed several guidelines and other resources dealing with the issues raised by obsolete pesticides. These are:

1. *The International Code of Conduct on the Distribution and Use of Pesticides* (FAO, 1986 and 1990).
2. *Disposal of Bulk Quantities of Obsolete Pesticides in Developing Countries* (FAO, 1996).
3. *Prevention of Accumulation of Obsolete Pesticide Stocks* (FAO, 1996).
4. *Pesticide Storage and Stock Control Manual* (FAO, 1996).

5. *Guidelines for the Management of Small Quantities of Unwanted and Obsolete Pesticides* (FAO, 1999).
6. *Assessing Soil contamination: A Reference Manual* (FAO, 2000).
7. *Baseline Study on the Problem of Obsolete Pesticide Stocks* (FAO, 2001).
8. A series of videos on disposal of obsolete stocks.
9. A website providing information on obsolete pesticide stocks.

http://www.fao.org/WAICENT/FAOINFO/AGRICULT/AGP/AGPP/Pesticid/Disposal/index_en.htm.

REFERENCES

1. FAO. *The International Code of Conduct on the Distribution and Use of Pesticides*; Food and Agriculture Organization, United Nations: Rome, 1986 and 1990.
2. FAO. *Disposal of Bulk Quantities of Obsolete Pesticides in Developing Countries: Provisional Technical Guidelines*; Food and Agriculture Organization, United Nations: Rome, 1996.

Oils

Deepak Raj Khajuria

Regional Horticultural Research Station, Dr. Y.S. Parmar University of Horticulture and Forestry, Kullu, Himachal Pradesh, India

Divender Gupta

Department of Entomology, Dr. Y.S. Parmar University of Horticulture and Forestry, Solan, Himachal Pradesh, India

INTRODUCTION

Oils are chemical substances composed essentially of carbon and hydrogen. Various types of oils such as petroleum oils, glyceridic fixed plant and fish oils, volatile or ethereal oils, and synthetic silicone and polybutene oils are being used to control a variety of pests in field and garden crops.^[1,2] Petroleum oil and neem oil have been in use against insects and mites for centuries. Today, with the availability of highly refined petroleum oil and neem oil safe for use on plants, there are renewed efforts to use oils in various integrated pest management (IPM) programs.^[3,4] In this chapter various technological and biological aspects of oils are discussed with respect to their use in pest management as winter and summer season applications.

WHY OILS FOR PEST CONTROL

Worldwide, more than 96,688 metric tons of pesticides were produced in 1995 of which 45% herbicides, 36% insecticides, and 17% fungicides were consumed.^[5] Despite the control of pests with synthetic pesticides, pests still remain a significant problem and are increasing in some crops. Arthropod pests and diseases incur 10–30% losses annually in various crops. Undoubtedly, the indiscriminate use of synthetic pesticides has increased the problem of insect resistance, environmental contamination, health hazards, and outbreaks of secondary pests, thus leading to their ban or regulation all over the world. Oils, on the other hand, are safe, eco-friendly, and biodegradable with minimal effects on the natural enemies. No species of insect has developed resistance to oils even after many decades of their continuous use. Oils therefore can safely be applied to complement chemical, biological, and cultural methods of pest control to combat insect resistance, environmental pollution, and the emergence of secondary pest problems in a variety of field and garden crops.

HOW OILS WORK

Oils produce a variety of effects on insects, mites, and fungal plant pathogens.^[6–8] Petroleum oils block respiratory holes (spiracles) through which insects and mites breathe, causing them to die of asphyxiation or suffocation. They may act as poisons by interacting with the acids and, eventually, the intercellular structures. Oils may also disrupt insect feeding, which is important in the transmission of some plant viruses by aphids. Ovipositional deterrent effects have also been observed in fruit flies and thrips. Petroleum oils also provide UV shield when used with biopesticides. Oils exhibit fungicidal and fungi-state action against fungal pathogens. They increase the resistance of the host plant by changing its physiology.^[1] Oils also act as carriers for ultra low volume (ULV) and low volume (LV) sprays. They are used as adjuvants, spreaders, and stickers in various pesticides to facilitate distribution, holding them to the leaf surface, resist weathering, and enhance pesticide absorption by the insects.

Vegetable oils (cottonseed, soybean, rapeseed, sunflower, groundnut, mustard, maize, etc.) and fish oils exhibit pesticidal, suffocating, antifeedant, and ovipositional deterrent effects. Another glyceridic oil extracted from the neem plant (*Azadirachta indica* A. Juss) contain biologically active substances such as azadirachtin and limonoids. Neem oils produce antifeedant, repellent, metabolic-inhibiting, toxicant, chemosterilant, ovipositional deterrent, and ovicidal effects on a variety of insects.^[4] They also possess good fungicidal properties. Similarly, chinaberry (*Melia azedarach* L.) oil produces neemlike effects and oil from pongram tree (*Pongamia pinnata* (L)) contains karanjin, which produces antifeedant, juvenile hormone analogues (JHA) and toxicant effects on insects. Volatile oils are of minor importance.

CHARACTERISTICS OF OILS FOR THEIR SAFE AND EFFICIENT USE

Oils are refined and then characterized for their distillation range, unsulfonated residue, viscosity, and

Table 1 Features of winter and summer management of pests with oils

| Winter season | Summer season |
|--|---|
| Oil application in dormancy (early dormancy—before bud swell to bud break stage and late dormancy—green tip to tight cluster stage) | Oil application in growing season (late spring to autumn) |
| Mostly on deciduous trees and rarely on evergreen trees | Both deciduous, and evergreen trees and other field crops |
| Single application as high-volume sprays | Mostly multiple applications as high-volume sprays |
| Oils used are: | Oil used are: |
| In early dormancy—heavy oils (lubricating oil emulsions or oils with >100 S viscosity, high boiling point and UR 50–90%) | Highly refined petroleum distillates supreme oils and summer oils used (<60 S viscosity, low distillation range, 50% distillation point, 212–224°C; 10–90% distillation range, 28°C; high UR value of >92% or more, high paraffinic contents) |
| In late dormancy—miscible oils (70–80 S viscosity, distillation range, 50% distillation point, 246°C; 10–90% distillation range, 29°C; UR 90–92% and moderate to high paraffinic contents) | |
| High conc. of oil applied (early dormancy >4.5% oil v/v, late dormancy—2 to 3% oil v/v) | Low conc. of oil used (0.5 to 1.0% oil v/v) |
| High mortality in overwintering stages only | Moderate to low mortality in eggs and sedentary or slow-moving, immature stages only |
| Complete coverage is easier | Complete coverage is difficult, oil deposit is important |
| Presence of synchronised life stages | All stages of insects in overlapping fashion |
| Safe to natural enemies | May be slightly toxic to the sedentary or slow moving stages of natural enemies |
| No conflict with other operations | Conflict with other operations |
| Least residual hazards | Residual hazards in certain high-cosmetic-value crops (grapes) |
| Injury mostly due to low temperatures (<40°F), freezing and high humidity | Injury due to high temperature (>100°F) with water stress or high humidity |
| Phytotoxicity in the form of burning of leaves, flower bud or twig drying, etc. | Phytotoxicity in the form of leaf burning, defoliation, fruit drop, or delayed fruit peel color, reduced total soluble sugars (TSS), etc. |
| Do not vary with plant type and cultivar | Vary with plant type and cultivar |

paraffinic character.^[1,2] Oils with high paraffinic contents are more pesticidal than the naphthenic oils. Unsulfonated residue (UR) in an oil indicates the amount of impurities. High UR oils (>90% UR) are safe to plants. Distillation range is the temperature at which 10%, 50%, and 90% of oil is distilled at low pressure of 10 mm Hg. If the distillation temperature for the 10–90% range is less, the oil has narrow range and is safe for use on plants. The mid-distillation temperature of a commercial oil is compared to that of pure paraffin with an identified number of carbon atoms. The commercial oils termed as C21 (415°C) and C23 (435°C) are based on mid-distillation point, but will also have lower and higher distillation compounds as per the 10–90% range. The viscosity of the oil is not of importance when the distillation range is given. After the oils have been characterized, they are emulsified with one or more emulsifiers to allow them mix with water and make an emulsion. Finally, these oils are available under different brand names as dormant oils, lubricating oil emulsions (heavy grade), mineral oils, miscible oils (medium grade), and superior

oils, supreme oils, and summer oils (light grade).^[6] Neem oil is extracted from seeds.^[7] The crude oil is purified to remove adulterants and to determine its color, odor, moisture, refractive index, iodine value, unsaponifiable matter, acid contents, viscosity, specific gravity, optical density, and aflatoxin contents. Oil extractions from chinaberry and pongram are similar to that of neem. Vegetable oils are processed to reduce the concentration of free fatty acids, phosphatides, iron, peroxides, and the odor.^[8]

OILS IN WINTER AND SUMMER MANAGEMENT OF PESTS

Oils as such are broad-spectrum pesticides that are phytotoxic. The efficacy of oils depends on the type of oil available, the host plant and its stage, and the insect and its stage where control is to be achieved. Environmental factors also affect pesticidal activity as well as the phytotoxicity of oil. Broadly, oils can

Table 2 Oils in winter and summer management of pests

| Name of the insect | Host plant | Type of oil/Trade name | Country (place) |
|--|--------------------------------|--|--|
| <i>Winter management</i> | | | |
| San Jose scale (<i>Quadraspidiotus perniciosus</i>) | Apple | In early dormancy Heavy oils, homemade/readymade lubricating oil emulsion Dormant Quick Mix Heavy [®] , Dormant Soluble [®] | USA (Ohio, Hawaii, California); India |
| Oystershell scale (<i>Lepidosaphes ulmi</i>) | Pear | Late dormancy Volcks [®] Sunspray [®] 6E or 7E HPSO [®] , SERVO [®] , ATSO [®] D-C-Tron [®] | USA (California, New York) India Australia |
| Rosy apple aphid (<i>Dysaphis plantaginea</i>) | Apple | Horticultural mineral oils | USA (California) |
| European red mite (<i>Panonychus ulmi</i>) | Pome | | USA (California, New York) Australia India |
| | Stone and nut fruits | | |
| Pear leafhopper (<i>Cacopsylla pyricola</i>) | Pear | | USA (California) |
| Pear mealy bug (<i>Planococcus</i> spp.) | Pear | | USA (California) |
| Brown mite (<i>Bryobia rubrioculus</i>) | Stone and nut fruits | | USA (California) |
| Leaf roller (<i>Archips agropyripila</i>) | Pome | | USA (California), India |
| | Stone and nut fruits | | |
| Tent hairy caterpillar (<i>Malacosoma americanum</i>) | Pome fruits | | USA (California) |
| Leaf curl aphid (<i>Brachycaudus helichrysi</i>) | Peach and stone fruits | | USA (California) |
| Terrapin scale (<i>Lecanium nigrofasciatum</i>) | Stone fruits | | USA (California) |
| Pine needle scale, striped scale, kermes scale, cottony maple scale (<i>Pulvinaria innumerabilis</i>) | Pome, stone and shade trees | Horticultural mineral oils | USA (California) |
| Leafhopper (<i>Erythroneura elagantula</i> , <i>E. variabilis</i>) | Grapes | Horticultural mineral oils | USA (California) |
| Citrus purple scale (<i>Lepidosaphes beckii</i>) | Citrus | Horticultural mineral oils | Italy |
| <i>Summer management</i> | | | |
| European red mite | Apple, pear | Volck Supreme Oil [®] , Sunspray [®] 6E plus, Sunspray Ultra Fine [®] , D-C-Tron Plus [®] | USA (New York, California), South Africa, Australia |
| San Jose scale | Apple | D-C-Tron [®] (nC21) | Australia |
| Woolly apple aphid (<i>Eriosoma lanigerum</i>) | Apple and pear | Sunspray Ultra Fine [®] , Volck Supreme [®] , Sunspray [®] | South Africa, USA |
| <i>Summer management</i> | | | |

(Continued)

Table 2 Oils in winter and summer management of pests (*Continued*)

| Name of the insect | Host plant | Type of oil/Trade name | Country (place) |
|---|--|--|---|
| Rosy apple aphid | Apple and pear | Sunspray Ultra Fine [®] , Volck Supreme [®] , Sunspray [®] | USA |
| Green apple aphid Summer Management (<i>A. pomii</i>) | Apple and pear | Sunspray Ultra Fine [®] , Volck Supreme [®] , Sunspray [®] | South Africa, USA |
| Spirea aphid (<i>Aphis spiraeicola</i>) | Apple and pear | Sunspray Ultra Fine [®] , Volck supreme [®] , Sunspray [®] | South Africa, USA |
| Codling moth (<i>Cydia pomonella</i>) | Apple and pear | Orchex 796 [®] | USA (Oregon, California) |
| | | Sunspray Ultra Fine [®] | South Africa |
| Two-spotted spider mite (<i>Tetranychus urticae</i>) | Stone and nut trees, strawberries | Horticultural mineral oils | USA (California) |
| Oblique-banded leaf rolle (<i>Christoneura, rosaceana</i>) | Apple | Horticultural mineral oils | USA (California) |
| Pacific spider mite (<i>T. pacificus</i>) | Stone and nut trees, and strawberries | Horticultural mineral oils | USA (California) |
| Peach silver mite (<i>Aculus cernutus</i>) | Stone and nut trees | Horticultural mineral oils | USA (California) |
| Navel orange born (<i>Ameylois transitella</i> Walker) | Stone and nut trees | Horticultural mineral oils | USA (California) |
| Pear psylla (<i>P. pyricola</i>) | Pear | Horticultural mineral oils | USA |
| Rust mite (<i>Aculus</i> spp.) | Pear | Horticultural mineral oils | USA |
| Blister mite (<i>Eriophyes pyri</i>) | | | |
| Pear mealy bug (<i>Planococcus</i> spp.) | Pear | | USA |
| Tea scale (<i>Aspidiodus</i> spp.) | Tea | Murphoil [®] | Kenya |
| Citrus red scale (<i>Aonidiella aurantii</i>) | Citrus fruits | Refined petroleum distillates | USA (California, Texas, Florida) |
| | | Sunspray [®] 7E, FC-435-66 [®] | |
| | | Petroleum oil concentrates | Italy, South Africa, Vietnam |
| | | D-C-Tron Plus [®] (nC23) | |
| Citrus red mite (<i>P. anonychus citri</i>) | Orange and lemon | Refined petroleum oils, Mineral oils, D-C-Tron Plus [®] (nC23) | USA (California), Japan, Malaysia, Vietnam and China |
| Citrus rust mite (<i>phyllocoptruta oleivora</i>) | Orange | F C-435-66 [®] , D-C-Tron Plus [®] (nC23) | USA (Florida), Malaysia, Vietnam |
| Bud mite (<i>Aceria sheldoni</i>) | Citrus fruits | F C-435-66 [®] , D-C-Tron Plus [®] (nC23) | USA (Florida), Malaysia, Vietnam |
| Web-spinning mite | Citrus fruits | F C-435-66 [®] , D-C-Tron Plus [®] (nC23) | USA (Florida), Malaysia, Vietnam |
| Arrowhead scale and white scale (<i>Unaspis yanonensis</i>) | Citrus fruits | Petroleum distillates | Japan (Kagawa, Nagashaki, Kyota) |
| Citrus purple scale (<i>L. beckii</i>) | Citrus | D-C-Tron Plus [®] | Australia (NSW) |
| White louse scale (<i>U. citri</i>) | Citrus | D-C-Tron Plus [®] | Australia (NSW) |
| Citrus leaf miner | Citrus | D-C-Tron Plus [®] | Australia, Malaysia |

(Continued)

Table 2 Oils in winter and summer management of pests (*Continued*)

| Name of the insect | Host plant | Type of oil/Trade name | Country (place) |
|--|------------|--|------------------------|
| Citrus mealy bug (<i>Planococcus citri</i>) | Citrus | D-C-Tron Plus [®] | China, Vietnam, Sicily |
| Chaff scale (<i>Parlatoria pergandi</i>) | Orange | Petroleum oils | USA (Texas) |
| Soft scale (<i>Ceroplastes floridensis</i>) | Citrus | Petroleum oils | Israel |
| Citrus aphid | Cotton | Mineral oils | USA (California) |
| Silver leaf white fly | Cotton | Mineral oils | USA (California) |
| Black scale (<i>Saissetia oleae</i>) | Citrus | Petroleum oils Sunspray [®] 7E | Portugal, Italy USA |
| Citrus white fly, citrus black aphid, citrus psylla | Citrus | PSO (nC24) | Malaysia |

be used for insect and mite control as winter and summer season applications^[9–11] (Table 1).

In winter a single application of oils is used as high-volume sprays, with heavy grades at higher concentrations (>4% oil v/v) in early dormancy and with lighter grade oils at lower concentrations (2–3% oil v/v) in delayed dormancy against the overwintering stages of insects and mites mostly on deciduous trees and rarely on evergreen trees^[1,4]. Delayed dormant applications are more effective as high mortality is achieved. Applications in winter are easier to apply, are safe to the natural enemies and have low or no residue hazards. In summer or the growing season, repeated applications with highly refined petroleum distillates are done at low concentrations (0.5–1% oil v/v) as high volume sprays on all kinds of tree and crops except for the oil sensitive plants.^[14] In the growing season, moderate to low mortality is achieved and chances of toxicity to natural enemies are higher. Oils provide direct control of certain pests and supplementary control to others. Thus, oils should be used in IPM packages with other components such as cultural practices and biological control.

EXAMPLES OF OILS IN PEST CONTROL

Petroleum and neem-based oils are used extensively against a wide range of insects, mites, diseases, and weeds (Tables 2–4). The role of petroleum oils in agriculture multiplied only after the discovery of kerosene oil soap emulsion in 1877 by a Michigan agriculture experiment station.^[2] By 1904, the first commercial miscible oil was in the market. From 1919 to 1923, lubricating oil emulsions were used as single, early dormant season applications against San Jose scales and aphids. Today, Dormant Soluble[®] and Dormant Quick Mix Heavy[®] oils are still popular and are

applied before bud break in California U.S.A., against scales, leaf curl aphids and mites in stone and nut fruits.^[3] In 1923, delayed dormant application of 2% lubricating oils provided very high mortality against San Jose scale when used with miscible oils. This application with oils is still widely employed to control scales, mites, hoppers, mealy bugs, caterpillars, and aphids in pome, stone, and nut fruits for higher economic returns with Sunspray[®] 7E, Volck Supreme Oil[®], unclassified petroleum oils in the United States, D-C-Tron[®] in Australia, and HPSO[®], SERVO[®], and ATSO[®] in India.^[9,12] Delayed dormant oils mixed with organophosphate insecticides are also used to enhance control. In order to supplement winter control, two to three applications of Volks Supreme[®], Sunspray[®] 6E Plus, Orchex 796[®] 786, or Sunspray Ultra Fine[®] in the United States, D-C-Tron Plus[®] (C23) in Australia, and Sunspray Ultra Fine[®] in South Africa are applied to European red mites, aphid complex on apples, pear pyrilla, mealy bugs, rust and blister mites on pears.^[3,10,11] Summer application is also used as a tactic approach to reduce red mite populations so as to enable its phytoseiid predator to control it effectively. Recently two to three applications of Orchex 796[®] along with mating disruption technique reduced 75% pesticide and the cost of protection by \$ 550/ha in Oregon, U.S.A.^[13] Citrus is another fruit that consumes a large amount of petroleum oils (3.4 million kg/yr, California, U.S.A., 1995). In citrus, product specifications and timing of sprays based on agroclimatic zones and sensitivity of citrus, varieties (lemon, grapefruit, Valencia orange, navel orange, and lime) have been developed and practiced in California.^[2] High-volume sprays of Sunspray[®] 7E in the United States, D-C-Tron Plus[®] in Malaysia, Vietnam, and China, and refined petroleum oils in South Africa, Italy, and Japan are used against red scale and mites in citrus for higher profits.^[14,15,19] Summer oils are also used to control

Table 3 Glyceridic oils (vegetable oils and plant oils) in pest control

| Name of the insect | Host plant | Type of oil/Trade name | Country (place) |
|--|-----------------------|--|------------------|
| <i>Vegetable Oils</i> | | | |
| San Jose scale | Apple | Degummed soybean oil Dormant sprays | USA (Tennessee) |
| Terrapin scale (<i>Mesolecanium nigrofasciatum</i>) | Apple, peach | | USA (Tennessee) |
| European red mite | Apple, peach | | USA (Tennessee) |
| White peach scale (<i>Pseudaulacuspis pentagona</i>) | Pome and stone fruits | Degummed soybean oil summer sprays | USA |
| <i>Aphis</i> spp. | Pome and stone fruits | | USA |
| Spirea aphid (<i>A. spiraecola</i>) | Pome and stone fruits | | USA |
| Cotton boll weevil (<i>Anthonomas grandis</i>) | Cotton | Soybean, cottonseed oil | Argentina |
| Citrus leaf miner (<i>Phyllocnistis chinensis</i>) | Citrus | Rapeseed oil (0.5%) | Australia |
| <i>Callosobruchus chinensis</i> , <i>C. maculatus</i> | Cowpea seeds | Castor oil, mustard oil, soybean oil, coconut, sunflower | India |
| <i>Plant oils (neem, chinaberry, pongram)</i> | | | |
| Citrus aphid (<i>Toxoptera aurantii</i>) | Citrus | Toxicant | India |
| Citrus psylla (<i>Diaphorina citri</i>) | Citrus | Repellent and reduced oviposition | India |
| Citrus black fly (<i>Aleurocanthus woglumi</i>) | Citrus | | India |
| Citrus white fly (<i>Bemisia tabaci</i>) | Citrus | | India |
| Citrus leaf miner (<i>P. citrella</i>) | Citrus | | India |
| Citrus red scale | Citrus | Toxicant (P) | India |
| Lemon butterfly (<i>Papilio demoleus</i>) | Citrus | Repellent | India |
| American bollworm (<i>Helicoverpa</i> spp., <i>H. armigera</i>) | Cotton, chickpea | Ovipositional deterrent | India |
| Red cotton bug (<i>Dysdercus koenigii</i> Fab.) | Cotton | Ovicidal effect | India |
| Spotted bollworm (<i>Earias vittella</i>) | Cotton | Ovicidal effect | India |
| Cotton white fly (<i>Bemisia tobaci</i>) | Cotton | Toxicant | India |
| Cotton aphid | Cotton | Toxicant | India |
| Pink bollworm (<i>Pectinophora gossypiella</i>) | Cotton | Growth inhibitory effects | India |
| Pear sawfly (<i>Caliroa cerasi</i>) | Pear | Antifeedant, metabolic inhibitor | Canada |
| Tobacco caterpillar (<i>Spodoptera litura</i>) | Polipagus | Antifeedant Ovicidel (P) | Canada, India |
| Desert locust (<i>Schistocerca gregaria</i>) | Polyphagous | Repellant, antifeedant | India |
| Migratory locust (<i>Locusta migratoria</i>) | | Repellent, antifeedant (C) | India |
| Leaf beetle (<i>Leptinotarsa decemlineata</i>) | | Ovipositional deterrent | India, Australia |

(Continued)

Table 3 Glyceridic oils (vegetable oils and plant oils) in pest control (*Continued*)

| Name of the insect | Host plant | Type of oil/Trade name | Country (place) |
|---|--------------|--|-----------------|
| Termites (<i>Macrotermes</i> spp.) | | Contact toxicity | India |
| Stem borer (<i>Chilo partellus</i>) | | | India |
| Hairy caterpillar (<i>Amsacta moorii</i>) | | | India |
| Brown plant hopper (<i>Nilaparvatha lugens</i>) | Rice | Repellent + chemosterilant, antifeedant | Kenya |
| White-banded plant hopper (<i>Sogatella furcifera</i>) | Rice | Repellent | |
| Earcutting caterpillar (<i>Mythmina separata</i>) | Rice | Repellent | |
| Rice leaf folder (<i>Cnaphalocrocis medinalis</i>) | Rice | Repellent | |
| Green leafhopper (<i>Nephotettix virescens</i>) | Rice | Repellent | India |
| <i>Plant oils (neem, chinaberry, pongram)</i> | | | |
| Bruchids (<i>Callosobruchus</i> spp.) | Stored grain | Ovipositional deterrent (0.75 ml oil) | India |
| Rice moth (<i>Corcyra cephalonica</i>) | Stored grain | Ovicidal effect | India |
| Leaf beetle (<i>Epilachna varivestis</i>) | Vegetables | Chemosterilant (C, N, P) | India |
| Mustard aphid (<i>Lipaphis erysimi</i>) | Vegetables | Toxicant | India |
| Pea aphid (<i>Rhopalosiphum nymphae</i>) | Vegetables | | India |
| Cucumber leaf miner (<i>Liriomyza trifolii</i>) | Vegetables | | India |
| Diamond black moth (<i>Plutella xylostella</i>) | Vegetables | Growth-inhibitory effects | India |

Note: Above effects in the table are for Neem (N). Effects of pongram and chinaberry are marked with P and C, respectively.

citrus rust mite, arrowhead scale, citrus leaf miner, soft scale, olive scale, rust mite, aphids, thrips, and mealy bugs.^[16,17] In cotton, vegetables, and ornamental crops oil applications are made against soft-bodied insects. Sprays of degummed soybean oil have successfully controlled apple insects. In India, application of Nimbecidine[®] (0.05%) has recorded a 28.8% increase in cotton yields as compared to 87.1% with endosulfan, and with neem oil 17.2% increase in yield as compared to 24.4% with monocrotophos. Similarly, Repelin[®], Neemark[®], and Ind-Ne[®] have also been used effectively in cotton. Neem oils are also used against citrus, vegetable, stored grain, and greenhouse pests in India and the United States. Similarly, pongram and chinaberry oils have also been found effective alone and in combination with neem oils.

Oils are also used against citrus greasy spot, Sigatoka, a disease of bananas, and certain aphid-transmitted viruses (Table 4).

CHALLENGES IN THE USE OF OILS FOR PEST CONTROL

Oils, a valuable tool in pest management systems, should be judged from their merits and properties that are different from the conventional pesticides. Major limitations in the use of petroleum oils in pest control include their low pesticidal efficiency, phytotoxicity, sensitivity to the environment (low and high temperature and high humidity), and various technological challenges associated with their refining and formulation (high paraffinic characteristics). Vegetable oils a renewable source are least exploited with regard to their refining techniques for making them safe to plants as well as for enhanced pesticidal activity.

Similarly, various obstacles limiting the use of neem oil include the lack of characterization of neem plant ecotypes for different environmental conditions, variations in neem formulations, poor shelf-life,

Table 4 Oils in disease control

| Name of the disease | Host plant | Type of oil |
|---|----------------------------------|--|
| Powdery mildew of apple (<i>Podosphaera leucotricha</i>) | Apple | Oil + baking soda (D-C-Tron Plus [®] , Australia) |
| Apple scab (<i>Venturia inaequalis</i>) | Apple | Orchex 796 [®] USA |
| Powdery mildew of cherry (<i>P. clandestina</i>) | Cherry | Stylet Oil [®] USA, Orchex 796 [®] |
| Powdery mildew of grape (<i>Uncinula necator</i>) | Grapes | Stylet Oil [®] , Sunspray Ultra Fine [®] USA |
| Bunch rot (<i>Botrytis cinerea</i>) | Grapes | Stylet Oil [®] USA |
| Downy mildew (<i>Plasmopara viticola</i>) | Grapes | Stylet Oil [®] USA |
| Powdery mildew of vegetables (<i>Sphaerotheca fusca</i>) | Muskmelon, squash, pumpkin | Sunspray Ultra Fine [®] USA, Canola oil |
| Tomato powdery mildew (<i>Leveillula taurica</i>) | Tomato | D-C-Tron Plus [®] |
| Powdery mildew of rose (<i>S. pannose</i>) | Rose | Sunspray Ultra Fine [®] USA |
| Black spot rose (<i>Diplocarpon. rosae</i>) | Rose | Sunspray Ultra Fine [®] USA |
| Greasy spot of citrus (<i>Mycosphaerella citri</i>) | Citrus | Refined petroleum distillates |
| Sigatoka diseases of banana (<i>M. musicola</i> <i>M. fijiensis</i>) | Banana | Oils, USA (Florida) |
| Citrus black spot (<i>Guignardia citricarpa</i>) | Citrus | Petroleum Oil |
| <i>Penicillium</i> spp. | Postharvest | Glyceridic Oils |
| <i>Aspergillus</i> spp. | Postharvest | Glyceridic Oils |
| Lily symptom less virus | Lilium | Sunspray [®] 11E |
| Lily mottle virus | Lilium | Luxan Oil-H [®] |
| Tospovirus | Tomato | D-C-Tron [®] (nC24) |
| Phytoplasma disease | Tomato | D-C-Tron [®] (nC24) |
| Celery mosaic virus | Celery | D-C-Tron [®] (nC24) |

phytotoxicity, wide variations in recommended doses, slow action and limited persistence, moderate mortality (as a result, required degree of control is not achieved), and difficulty in enriching the azadirachtin contents in neem oil above that present in seed kernels. In addition, standardization and investigation of various compounds present in it and the absence of aflatoxin in oil is difficult to ensure.^[7,18]

FUTURE PROSPECTS

There has been tremendous advancement in chemistry, refinement, and diversification in the use of various types of oils in pest control. Today, oils are used in direct and supplementary control of insects and mites. The potential of petroleum and neem oils can be judged from the diversity of crops to which they are applied and the number of crops for which specific guidelines for their use against insects, mites, and diseases have been established.^[3] Yet their use is limited to certain crops and their pests.

To improve their applications on these crops more and more research efforts are required to make oils

safe to plants and effective against pests. Similarly, vegetable oils have also shown promise as a safe pesticide. There are committed teams of researchers in various parts of the world who continue to develop technologies to improve their safety to the host plants and enhanced pesticidal activity to the pests. More and more crops are being brought under the use of various types of oils worldwide. Currently, different types of oils are being registered for their use on different crops. All oils are safe, inexpensive, and biodegradable; therefore they could play an important role in the development of future IPM systems that rely more on safe options and less on conventional pesticides.

REFERENCES

1. Northover, J.; Timmer, L.W. Control of plant diseases with petroleum and plant oils. In *Spray Oils Beyond 2000 Sustainable Pest and Disease Management*, Sydney, Australia, Oct 25–29, 1999; Beattie, G.A.S., Watson, D.M., Eds.; University of Western Sydney Hawkesbury and University of Florida: Sydney, 1999; Polym. Mater. Sci. Eng., 47.

2. Metcalf, C.L.; Flint, W.P. Insect control. In *Destructive and Useful Insects*; McGraw Hill Company, Inc.: New York, 1962; 366 pp.
3. Zalom, F.G.; Walsh, D.B. Horticultural spray oils; A useful tactic in California IPM system. *Polym. Mater. Sci. Eng.*, 78. The paper has been presented in *Spray oil 2000 in Australia*.
4. Dhaliwal, G.S.; Arora, R. Botanical pesticides. In *Principles of Insect Pest Management*; National Agricultural Technology Information Center: Ludhiana, 1996; 147–174.
5. Nigam, G.L.; Murthy, K.S. An optimum use of pesticides in IPM Technology. *Pestic. Inf.* **1999**, 25 (4), 6–9.
6. [http://www.yahoo.com/Horticultural oils in Arthropod pest management](http://www.yahoo.com/Horticultural_oils_in_Arthropod_pest_management) (accessed November 2000).
7. Gahukar, R.T. Commercial and industrial aspects of neem based pesticides. *Pestology* **1998**, 22 (10), 15–41.
8. Deyton, D.E.; Sams, C.E.; Pless, C. Soyabean oil delays peach flowering, thins fruits and kills key arthropod pests of deciduous fruit trees. *Polym. Mater. Sci. Eng.*, 58. The paper has been presented in *Spray oil 2000 in Australia*.
9. Lawson, D.S.; Weires, R.W. Management of European red mite (Acari; Tetranychidae) and several aphid species on apple with petroleum oils and an insecticides soap. *J. Econ. Entomol.* **1991**, 84 (5), 1550–1557.
10. Agnello, A.M.; Reissig, W.H.; Harris, T. Management of summer populations of European red mite (Acari: Tetranychidae) in apple with horticultural oils. *J. Econ. Entomol.* **1994**, 87 (1), 148–161.
11. Thwaite, W.G.; Eslick, M.A.; Nicol, H.I. Evaluation of petroleum spray oil for apple integrated pest and disease management. *Polym. Mater. Sci. Eng.*, 72. The paper has been presented in *Spray oil 2000 in Australia*.
12. Bhalla, O.P.; Gupta, P.R. Insect pests of temperate fruits. In *Advances in Horticulture: Fruit Crop*; Chadha, K.L., Pareek, O.P., Eds.; Malhotra Publishing House: New Delhi, 1993; Vol. 3, 1557–1589.
13. Van Buskirk, P.; Hilton, R.; Riedl, H. Use of narrow range petroleum spray oil for suppression of codling moth and secondary arthropod pests in an area wise mating disruption program. *Polym. Mater. Sci. Eng.*, 73. The paper has been presented in *Spray oil 2000 in Australia*.
14. Botha, J.H.; Plessis, D.; Calitz, F.J.; Du Plessis, D. Effect of Sunspray Ultra Fine® summer oil on European red mite in apple orchards. *J. S. Afr., Soc. Hortic. Sci.* **1995**, 5 (2), 101–105.
15. Leong, S.C.T.; Ng, H.L.; Beattie, G.A.C.; Watson, D.M. Comparison a petroleum spray oil and conventional pesticides for control of major citrus pests, Sarawak, Malaysia. *Polym. Mater. Sci. Eng.*, 62. The paper has been presented in *Spray oil 2000 in Australia*.
16. Nguyen, V.C. Development of petroleum spray oil and natural enemy based integrated post management programs for citrus in northern Vietnam. *Polym. Mater. Sci. Eng.*, 66. The paper has been presented in *Spray oil 2000 in Australia*.
17. Ping, L.; Piao, Y. Citrus pests in China and their sustainable management. *Polym. Mater. Sci. Eng.*, 75. The paper has been presented in *Spray oil 2000 in Australia*.
18. Dhawan, A.K. Potential of neem in cotton pest management in India: an update. In *Green Pesticides, Crop Protection and Safety Evaluation*; Agnihotri, N.P., Walia, S.C., Gaj bhiye, V.T., Eds.; Society of Pesticide Science: New Delhi, 1999; 62–75.
19. Riehl, L.A. Update, 1981–1987, of developments in mineral spray oils. Proceedings of the Sixth International Citrus Congress, Tel Aviv, Mar 6–11, 1988; Goren, R., Mendel, K., Eds.; Balaban Publishers Rehovot: Israel, 1998; 1253–1267.

Olive Insects: Ecology and Control

Marshall W. Johnson

Department of Entomology, University of California-Riverside, Riverside, California, U.S.A.

Kent M. Daane

Department of Environmental Science, Policy, and Management, Kearney Agricultural Center, University of California-Berkeley, Berkeley, California, U.S.A.

INTRODUCTION

Olives have been cultivated and their products traded in the Middle East for more than 5000 years.^[1] Because olives do best when summers are long and hot and winters relatively cool ($>9^{\circ}\text{C}$), worldwide commercial olive production is confined to latitudes between 30° North and 45° South. The Mediterranean basin is the largest olive production area, where most olives are grown for oil for human consumption and industrial use.^[2] As of 2002, the leading producers (in order) of oil and table olives (combined) were Spain, Italy, Greece, Turkey, Syria, Morocco, Tunisia, and Portugal. Olives are also produced in other areas including South Africa, South America, Australia, and California in the U.S.A.

Although greater than 125 arthropod species attack olive plants, the olive fly, *Bactrocera oleae* Gmelin (Diptera: Tephritidae) (Fig. 1), is the major threat to olives worldwide.^[3] In table olives, the larval damage is largely cosmetic but can also increase rot, and only minimal infestations are tolerated. Acceptable levels of damage in olives destined for oil production are higher (about 10%).^[3] Less important pests attacking olives include Lepidoptera: olive moth, *Prays oleae* (Bernard), jasmine moth, *Palpita unionalis* Hübner, olive pyralid moth, *Euzophera pinguis* Haworth, and leopard moth, *Zeuzera pyrina* L.; Homoptera: black scale, *Saissetia oleae* (Olivier), olive scale, *Parlatoria oleae* Colvée, and oleander scale, *Aspidiotus nerii* (Bouche); and Coleoptera: olive bark beetle, *Phloeotribus scarabaeoides* Bern, and twig cutter beetle, *Rhynchites cribripennis* (Desbrocher des Loges), as well as the olive thrips, *Liothrips oleae* Costa, and olive psylla, *Euphyllura olivina* Costa.^[3-6] The importance of these pests varies with location, climate, and the intended use of the olives.

The following discussion focuses on managing the olive fly, olive moth, black scale, and olive scale—the most commonly encountered olive pests. Although conventional pesticide treatments may be applied for all, the most effective controls for olive fly and the scale species have resulted from the development of control

measures that rely on a better understanding of the insects' biology and ecology.

THE OLIVE FLY

Life History

The adult olive fly is about 4.5 mm in length (Fig. 1) and lives from two to six months depending upon food availability and temperature.^[4] Foods eaten by adult flies may include honeydew, rotting fruits, and bird feces. Although olive fly only reproduces on olive fruit, the adults commonly disperse to surrounding vegetation (e.g., citrus, walnuts). Eggs are laid under the skin of the olive fruit and are difficult to detect. One fly may lay up to 500 eggs.^[4] There are three larval stages (i.e., instars). Population densities can vary greatly with season and temperature. High summer temperatures ($>38^{\circ}\text{C}$) can cause high adult mortality if access to food and water is lacking (M.W. Johnson, unpublished data).

Monitoring

One key to effective management is routine assessment of pest densities to determine the need for management actions. Unless an orchard is highly infested, olive flies are difficult to monitor in the fruit. Fortunately, adult flies may be trapped using either yellow, sticky panel traps or McPhail traps (glass or plastic) (Fig. 2) that are baited with attractive compounds.^[4] For panel traps, a food lure (e.g., ammonium bicarbonate or ammonium carbonate) and a synthetic male sex lure (spiroketal) are usually attached to the trap. McPhail traps commonly employ *Torula* yeast and borax (stabilizer) dissolved in water to attract flies.

Management Options

The intended use of harvested fruit determines the acceptable level of olive fly infestation. Olives destined for pressing can tolerate higher infestations (10% or



Fig. 1 Female olive fly laying egg on olive fruit.

more) than those olives intended for curing (near zero). Many European countries have government-sponsored management programs that provide area-wide spray programs. Average crop losses using current control measures in Europe vary between 5% and 15%.^[3] In other locations (e.g., California), individual growers are responsible for their control costs and control actions.

The standard control method in many places is the use of insecticidal bait sprays. These consist of a bait (e.g., chemical or enzymatic protein hydrolyzates, ammonia releasing salts, urea, or microencapsulated sex attractant) and a small dose of insecticide (e.g., Spinosad[®]).^[3] Insecticidal baits must be ingested to kill flies. Olive orchards must be sprayed frequently (e.g., weekly) with insecticidal baits to maintain low fly populations during the warmer months. Area-wide



Fig. 2 Plastic McPhail trap used to monitor olive fly adults.

spray programs are essential for the best control using bait sprays.^[3]

Conventional cover sprays (i.e., treatment of all foliage with diluted insecticide) using organophosphates and other insecticides are still used in Europe and other locations.^[3] They are recommended when bait sprays have failed to reduce high olive fly populations. Their use is typically limited to emergency situations because of adverse environmental side-effects.

“Attract and Kill” traps may also be employed in which food and male sex lures are used to entice adult flies to land on an insecticide-impregnated substrate. Flies that land on the trap pick up a lethal insecticide dose and die shortly thereafter. The advantage of this method is that the traps can remain effective for months, and non-target impacts are low.

Management Prospects

Efforts are underway in Europe and California to improve the effectiveness of olive fly biological control. In both locations, experimental efforts are underway to develop and refine mass releases of parasitoids (i.e., augmentation) for short-term control of larvae. Attempts to introduce exotic natural enemies to California from Hawaii, Europe, and Africa are in progress.

THE OLIVE MOTH

The olive moth has a more limited distribution than the olive fly and is found from the Mediterranean basin to the northeastern shores of the Black Sea.^[6] Unlike the olive fly, the olive moth infests flowers, fruit, and leaves (based on the overlap of specific generations with the phenological stages), and this results in a reduction of fruit set, increased fruit drop, and overall weakening of the tree, respectively.^[3,6] High population densities may cause economically significant crop losses. Control methods (conventional insecticide sprays or dusts) are usually applied to the generation attacking the fruit.^[3] Control of the spring generation with conventional insecticides is avoided because of their catastrophic effects on the beneficial fauna of olive orchards, which are highly active in spring. Alternative control methods are available. *Bacillus thuringiensis* Berliner is effective against the generation attacking flowers,^[6] while chitin synthesis inhibitors (e.g., triflumuron) are effective against the generation attacking fruit.^[3]

BLACK SCALE

Black scale (Fig. 3) is native to southern Africa but has spread throughout the world's olive production



Fig. 3 Black scale immatures and adults on olive stem.

areas.^[4] The pest usually has one generation per year, but two generations are possible when conditions permit. It feeds on plant juices by inserting its stylets into leaves and twigs, and excretes excess ingested plant materials as honeydew. Honeydew that accumulates on foliage promotes fungus development (i.e., sooty mold), which can potentially reduce leaf respiration and photosynthesis. Black scale feeding and honeydew/sooty mold accumulation can decrease fruit bud formation, induce leaf drop and twig dieback, and reduce crop yield.^[4] When infestations are high, economic damage occurs. Black scale is attacked by several parasitoids [e.g., *Metaphycus helvolus* Compere, *M. hageni* Daane and Caltagirone, *Scutellista caerulea* (Fonscolombe)] and predators (e.g., green lacewings, ladybeetles). Their effectiveness varies with climatic area. Although pesticides (e.g., oils, organophosphates, carbamates) can control populations, an effective cultural control is available. In some areas, pruning of the interior tree canopies will increase canopy temperatures, causing immature black scale stages to desiccate and die when summer temperatures surpass 38°C.^[4] A combination of canopy pruning with effective biological control agents is an excellent way to reduce the need for chemical treatments.

OLIVE SCALE

Olive scale has a cosmopolitan distribution and may be found in Argentina, India, the Mediterranean, Middle East, Russia, Turkey, and California.^[4] As with black scale, high olive scale densities may result in tree defoliation and twig death and frequently reduce crop yield. Major damage results when dispersing scale crawlers settle on fruit.^[4] When fruit are infested by the first generation of the season, fruit become badly

misshapen. Infestation by the second generation results in purple spotting of green fruit, rendering them unmarketable. In California, effective control was achieved by the classical introduction of two parasitoid species, *Aphytis maculicornis* (DeBach and Rosen) and *Coccophagoides utilis* (Doutt).^[7] These parasitoids work in unison to suppress olive scale over a wide range of climatic conditions. Currently, chemical controls for olive scale are infrequently needed unless the biological control agents are disturbed by pesticide treatments applied for other pests.^[4]

CONCLUSIONS

The olive fly continues to be the primary pest of olives in most olive production areas. Development and refinement of more suppression tactics that take advantage of the fly's behavior (e.g., insecticide baits, attract and kill traps) hold the promise for greater effectiveness with reduced pesticidal inputs. Continuing efforts to discover and use parasitoids as control agents of olive fly hold some hope for production areas where the climatic conditions permit survival and reproduction of natural enemies.

REFERENCES

1. Connell, J.H. History and scope of the olive industry. In *Olive Production Manual*, 2nd Ed.; Sibbett, G.S., Ferguson, L., Coviello, J.L., Lindstrand, M., Eds.; University of California Agriculture and Natural Resources Publication 3353: Oakland, CA, 2004; 1–13.
2. Abassi, F., Ed.; The world olive oil market, the world table olive market. *Olivae* **2002**, *10* (92), 22–27.
3. Haniotakis, G.E. Olive pest control: present status and prospects. In *Proceed. 1st European Meeting of the IOBC/WPRS Study Group on Integrated Protection of Olive Crops*, Ghania, Crete, Greece, May 29–31, 2003, IOBC/WPRS Bulletin.
4. Daane, K.M.; Rice, R.E.; Zalom, F.G.; Barnett, W.W.; Johnson, M.W. Arthropod pests of olive. In *Olive Production Manual*, 2nd Ed.; Sibbett, G.S., Ferguson, L., Coviello, J.L., Lindstrand, M., Eds.; University of California Agriculture and Natural Resources Publication 3353: Oakland, CA, 2004; 105–114.
5. Arambourg, Y. The olive's entomological fauna: the main species entailing economic repercussions. *Olivae* **1984**, *2*, 39–44.
6. Arambourg, Y. The olive's entomological fauna: species of localized economic significance. *Olivae* **1984**, *4*, 14–21.
7. Huffaker, C.B.; Kennett, C.E. Studies of two parasites of olive scale, *Parlatoria oleae* (Colvée): IV. Biological control of *Parlatoria oleae* (Colvée) through the compensatory action of two introduced parasites. *Hilgardia* **1966**, *37* (9), 283–335.

Organic Soil Amendments

Philip Oduor Owino

Department of Botany, University of Kenyatta, Nairobi, Kenya

INTRODUCTION

Modern agriculture is faced with the challenge of becoming more productive and yet more sustainable. One important goal toward this end is to boost crop production through proper management of weeds, insect pests, and plant pathogens. These management tactics must be implemented without adversely affecting the ecosystem. Therefore, there is a need to change from the use of pesticides to safer pest management practices, which can be adopted in integrated pest management (IPM) programs. The use of organic soil amendments for the control of plant pathogens and/or pests may provide a viable alternative.^[1-3]

ORGANIC SOIL AMENDMENTS AND THEIR MECHANISMS OF ACTION

Amending soil with organic matter such as chitin, oil cakes, compost, animal manures, and other industrial by-products in pest management studies is well recognized.^[4-5] However, effects of these materials on disease development are not clear, and have been attributed, in part, to the factors discussed below.

Impacts of Organic Soil Amendments on Plant Health and Weeds

Soil amendments improve plant growth by enhancing plant nutrition.^[1] The levels of nitrogen, phosphorus, potassium, and other essential elements are increased when organic matter is added to soil and is associated with better crop performance.^[1] Changes in physical characteristics of soil may also enhance plant growth and the associated weeds, an attribute that should be utilized in disease management. Healthy plants produce higher yields, compete with weeds, and tolerate fungal, nematode, and insect damage better than unthrifty plants.^[1,6]

Organic Soil Amendments and Plant Resistance

Materials such as oil cakes and sawdust have high phenolic content and alter the attractiveness of host plants to nematodes.^[7] For example, seed treatment with

ground oil cakes boosts plant resistance to *Tylenchulus semipenetrans* Cobb and root-knot nematodes due to increased levels of phenols in treated citrus and tomato roots, respectively.^[4,7] In contrast, organic materials from *Tithonia diversifolia* (Hems) and chicken manure increase the severity of dry root-rot of French bean (*Phaseolus vulgaris* L.cv. Monel) caused by *Fusarium solani* f.sp. *phaseoli* (Mart) Sacc. (Table 1). This has been attributed to the formation of stimulatory ammonium compounds during decomposition.^[8]

Release of Compounds Toxic to Insects and Plant Pathogens

Some organic materials release insecticidal, nematotoxic, and/or fungitoxic chemicals during decomposition, for instance, neem oil and neem cake powder from the neem tree, *Azadirachtin indica*. A. Juss contains the limonoid azadirachtin, which is nematotoxic and insecticidal in nature.^[6,9] The black bean aphid, *Aphis fabae* (Scop), has been successfully controlled by this product (Table 2). The nematocidal activity of marigolds (*Tagetes* spp.) and castor (*Ricinus* spp.) has also been recognized, but in this case the toxic principles are Polythienyls and ricin, respectively.^[4,7] Antimicrobial chemicals such as nitrites and hydrogen sulfide are also produced during decomposition and play an important role in disease control. Unfortunately, various changes in quality and quantity of these chemicals occur over time, making it difficult to obtain more than circumstantial evidence that any one compound is responsible for disease suppression.^[2]

Stimulation of Antagonistic Microorganisms

The hypothesis that organic soil amendments stimulate the activity of antagonistic microorganisms was proposed over 50 years ago.^[5] When organic matter is added to soil, a sequence of microbial changes is initiated, none of which should be viewed in isolation. It is possible that the ability of nematophagous fungi such as *Paecilomyces lilacinus* Thom. (Samson) and *Verticillium chlamydosporium* (Goddard) to destroy/parasitize eggs of root-knot nematodes is stimulated by soil amendments.^[2,10] Egg parasitism of up to 37%

Table 1 Effect of cowdung (Cd) and organic soil amendments from *T. diversifolia* (Td) and their combination with metalaxyl (Mt) on plant growth and dry root rot of French beans 72 days after planting in soils inoculated with *F. solani* f.sp. *phaseoli* (Fs)

| Soil treatment | Mean shoot dry weight (g) | Mean root dry weight (g) | Mean ^a L.D.E.T (mm) | Mean root rot index ^b (1–9) | Mean number pods per plant | Mean dry weight of 100 seeds |
|----------------|---------------------------|--------------------------|--------------------------------|--|----------------------------|------------------------------|
| Cd+Fs | 4.84b ^c | 1.398a | 30.0bc | 3.13c | 22.0a | 25.05a |
| Cd+Mt+Fs | 4.26b | 1.166b | 50.0abc | 5.41b | 21.0a | 19.02b |
| Td+Fs | 3.36b | 0.232b | 62.1ab | 6.23b | 5.0b | 23.57b |
| Td+Mt+Fs | 1.03c | 0.014e | 90.0ab | 8.00a | 0.0c | 0.00c |
| Fs alone | 3.12b | 0.167e | 97.5a | 7.25a | 4.0bc | 21.37a |
| +Cd; No Fs | 7.84a | 0.733c | 6.11c | 1.00d | 22.0a | 32.17a |
| Td alone | 2.31c | 0.796c | 5.43c | 1.00d | 4.0bc | 32.85a |

^aLength of discolored tissue (mm) (L.D.E.T).

^bMean root-rot index was based on a 0–10 rating scale, where, 0 = no symptoms and 10 = whole root system decayed.

^cNumbers are means of five replicates. Means followed by the same letter within the same column are not significantly different at $P = 0.05$ level by Duncan's Multiple Range Test (DMRT). (From Ref.^[14])

has been achieved with organic matter from castor plant or chicken manure (Table 3). Besides egg parasitism, the diverse range of microorganisms in amended soils competes with nematodes and other invertebrate pests for space and oxygen, thereby creating unfavorable anaerobic microsites in the soil. Bacteria such as *Streptomyces anulatus* (Beijerinck) Waksman, the collembolan, and *Entomobyroides dissimilis* (Moniez) are good examples.^[5] *Armillaria* root rot of fruit and forest trees, caused by *Armillaria mellea* Vahl ex.fr, is minimized using coffee pulp that stimulates the antagonistic effects of *Trichoderma viride* link ex. Fries against a wide range of *Armillaria* spp.^[11]

In conclusion, it is evident that various activities of soil microorganisms contribute significantly to the detrimental effects of organic matter on plant pathogens. However, it is difficult to determine whether any one activity or group of organisms is directly responsible for the suppression of specific diseases. The evidence available remains largely circumstantial.

USE OF ORGANIC SOIL AMENDMENTS IN THE 21st CENTURY

Studies on the efficacy of organic soil amendments against plant pathogens should be intensified worldwide. Organic plant materials such as chitin, compost, and oil cakes have great nematode control potential but have remained unutilized in biological control systems due to inadequate and inconsistent information on their efficacy and compatibility with antagonistic microorganisms.^[10,12] It is not known if these organic materials and fungal antagonists/predators can successfully be integrated into the same pest control systems. The future challenge in this case is to determine ways of boosting the antagonistic potential of specific beneficial organisms by using locally available amendments in quantities realistic for broad-scale agricultural use. The complexity of the soil environment may thwart efforts to achieve this, but previous studies^[4–5,7] and recent work on the interaction between

Table 2 Weekly mean aphid scores on French beans following treatments with the insecticide Gaucho Neem Kernel (NKCP) and different neem products

| Treatment | Week 1 | Week 2 | Week 3 | Week 4 | Mean no. of pods/plant (Week 4) |
|------------------|-------------------|--------|--------|--------|---------------------------------|
| Karate (2 ml/L) | 0.1c ^a | 0.1c | 0.6c | 1.2c | 1.03c |
| Neem oil EC (3%) | 0.1c | 0.5c | 0.5c | 1.3c | 9.3c |
| NKCP/WE (50 g/L) | 1.1b | 1.0c | 0.7c | 1.6c | 6.2c |
| Gaucho (8 ml/kg) | 0.9b | 1.5b | 2.1b | 3.6b | 5.0b |
| Control | 3.4a | 4.9a | 6.7a | 7.0a | 3.3a |

^aNumbers are means of 10 replicates. Means followed by the same letter within columns do not differ significantly at $P = 0.05$ by Duncan's Multiple Range Test (DMRT). (From Ref.^[6])

Table 3 Effect of organic soil amendments, and soil treatments with captafol or aldicarb on the parasitism (%) of *Meloidogyne javanica* eggs with *P. lilacinus* and growth of tomato cv money maker plants

| Soil treatment ^a | Egg parasitism (%) | Juveniles/300 ml soil | Gall index ^b (0–4) | Shoot height (cm) | Shoot dry weight (g) |
|-----------------------------|--------------------|-----------------------|-------------------------------|-------------------|----------------------|
| Tag+Mj | 0.5f | 188c | 2.3c | 36.0f | 2.8g |
| Dat+Mj | 0.7f | 189c | 2.5c | 35.0g | 2.9g |
| Ric+Mj | 1.2f | 207c | 2.4c | 32.1h | 3.5e |
| Ch.M+Mj | 1.0f | 217c | 1.8d | 46.8b | 5.0b |
| Ald+Mj | 0.8f | 18e | 1.3e | 49.8a | 5.4a |
| Cap+Mj | 0.0f | 521a | 3.8a | 32.5h | 2.9g |
| F+Mj | 21.2e | 438b | 3.5b | 28.4i | 1.7i |
| F+cap+Mj | 1.3f | 501a | 3.6ab | 30.2hi | 2.6h |
| F+Ald+Mj | 26.2d | 10e | 0.5f | 46.6b | 5.1b |
| F+Tag+Mj | 30.9b | 206c | 2.0d | 39.2e | 3.0g |
| F+Dat+Mj | 28.4c | 201c | 2.0d | 42.8c | 3.2f |
| F+Ric+Mj | 37.2a | 187c | 2.5c | 39.7e | 3.6e |
| F+Ch.M+Mj | 37.3a | 147d | 1.8d | 42.0d | 4.2d |
| Mj. “Only” | 0.5f | 425b | 3.4b | 23.5j | 1.4j |
| Soil “Only” | 0.0f | 0.0e | 0.0f | 44.9bc | 4.5c |

^aF = fungus; Mj = *M. javanica*; Cap = Captafol; Ald = aldicarb; Tag = *Tagetes minuta*; Dat = *Datura stramonium*; Ric = *Ricinus communis*; and Ch.M = chicken manure.

^bGall index was based on a 0–4 rating scale, where 0 = no galls and 4 = 76%–100% of the root system galled.

^cNumbers are means of 10 replicates. Means followed by different letters within a column are significantly different ($P = 0.05$) according to Duncan's Multiple Range Test.

(From Ref. [1].)

nematodes and organic soil amendments^[1–3,13] suggest that this is a promising area for further research.

FUTURE CONCERNS

Organic soil amendments have a positive future in pest and disease control.^[10,14] However, recent techniques used in the fields of biotechnology and molecular genetics^[15] may dominate biological control research with a view of alleviating problems that are presently confronting researchers in an attempt to look for safe pest control alternatives. It is important that scientists, in their eagerness to embrace these new technologies, do not lose sight of the fact that the ultimate objective is the development of environmentally friendly pest control systems that can be applied in the field. We must strike the right balance between theoretical investigations and the more applied biological control studies aimed at developing viable pest management options.

REFERENCES

- Oduor-Owino, P. Fungal parasitism of root-knot nematode eggs and effects of organic matter, selected agrochemicals and intercropping on the biological control of *Meloidogyne javanica* on tomato. In *Ph.D. thesis*; Kenyatta University: Nairobi, 1996.
- Oduor-Owino, P.; Waudo, S.W. Effects of delay in planting after application of chicken manure on *Meloidogyne javanica* and *Paecilomyces lilacinus*. *Nematol. Mediter.* **1996**, *24* (3), 7–11.
- Oduor-Owino, P.; Sikora, R.A.; Waudo, S.W.; Schuster, R.P. Effects of aldicarb and mixed cropping with *Datura stramonium*, *Ricinus communis* and *Tagetes minuta* on the biological control and integrated management of *Meloidogyne javanica*. *Nematologica* **1996**, *42* (2), 127–130.
- Bhattacharya, D.; Goswami, B.K. Comparative efficacy of neem and groundnut oil-cakes with aldicarb against *Meloidogyne incognita* in tomato. *Rev. Nematol.* **1987**, *10* (1), 467–470.
- Linford, M.B. Stimulated activity of natural enemies of nematodes. *Science* **1937**, *85* (1), 123–124.
- Maundu, M.E. Control of the black aphid, *Aphis fabae* scop of beans using neem-based pesticides in Kenya. In *M.Sc. thesis*; Kenyatta University: Nairobi, 1999.
- Bandra, T.; Elgindi, D.M. The relationship between phenolic content and *Tylenchulus semipenetrans* populations in nitrogen-amended citrus plants. *Rev. Nematol.* **1979**, *2* (3), 161–164.
- Waudo, S.W.; Oduor-Owino, P.; Kuria, M. Control of Fusarium wilt of tomatoes using soil amendments. *East Afr. Agric. For. J.* **1995**, *60* (4), 207–217.

9. Oduor-Owino, P.; Waudu, S.W. Comparative efficacy of nematicides and nematicidal plants on root-knot nematodes. *Trop. Agric.* **1994**, *71* (4), 272–274.
10. Oduor-Owino, P.; Waudu, S.W.; Sikora, R.A. Biological control of *Meloidogyne javanica* in Kenya. Effect of plant residues, benomyl and decomposition products of mustard (*Brassica campestris*). *Nematologica* **1993**, *39* (3), 127–134.
11. Onsando, J.M.; Waudu, S.W.; Magambo, M.J.S. A biological control approach to root rot of tea *Armillaria mellea* in Kenya. *Tea* **1989**, *10* (2), 165–173.
12. Oduor-Owino, P.; Waudu, S.W.; Makhatsa, W.L. Effect of organic amendments on fungal parasitism of *Meloidogyne incognita* eggs and growth of tomato (*Lycopersicon esculentum* Mill) cv money maker. *Int. J. Pest Manage.* **1993**, *39* (4), 459–461.
13. Oduor-Owino, P.; Waudu, S.W. Medicinal plants of Kenya. Effects of *Meloidogyne incognita* and the growth of Okra. *Afro-Asian J. Nematol.* **1992**, *2* (1), 64–66.
14. Wagichunge, A.G. Efficacy of seed-dressing and organic amendments against fusarium root-rot of french beans (*Phaseolus vulgaris* L.cv. Monel) in Kenya. In *M.Sc. thesis*; Kenyatta University: Nairobi, Kenya, 2000.
15. Kerr, A. Commercial release of a genetically engineered bacterium for the control of crown gall. *Agric. Sci.* **1989**, *2* (1), 41–44.

Ornamental Crop Pest Management: Plant Pathogens

D. Michael Benson

Department of Plant Pathology, North Carolina State University,
Raleigh, North Carolina, U.S.A.

INTRODUCTION

For ornamental crop diseases, integrated pest management (IPM) is a widely practiced approach for disease control because so many culture variables are involved in nursery production. Components of IPM for ornamental diseases in nursery production include cultural practices and sanitation, disease resistance, scouting, chemical and biological control. Because ornamentals are a high-value commodity in which epidemics can develop rapidly, nurserymen are well aware of the need to practice IPM. Major plant diseases include root rot and wilt caused by species such as *Phytophthora*, *Rhizoctonia*, *Fusarium*, and *Verticillium*; canker and dieback caused by species such as *Phomopsis*, *Botryosphaeria*, *Phytophthora*, and *Nectria*; leaf spot and blight caused by species such as *Cercospora*, *Entomosporium*, *Colletotrichum*, *Pseudomonas*, and *Erwinia*; powdery mildew caused by several fungal pathogens, as well as decline caused by plant-parasitic nematodes.

ORNAMENTAL CROP PRODUCTION

Woody ornamentals produced in nurseries may be container-grown or field-grown for periods ranging from a single season to several years. Soilless potting mix consisting of a high percentage of coniferous tree bark is used for container production. Soilless mixes are well drained and lightweight with good physical properties for optimizing plant growth. Containers arranged by plant size and watering needs are placed on a ground cover, such as fabric or gravel, to control weed growth and to prevent dispersal of soilborne pathogens from underlying soil. Field-grown ornamentals are produced in the ground over several growing seasons for the landscape market where large plants are needed to give a landscape design an instant, finished look.

Pathogens Groups that Attack Ornamentals

Fungal pathogens cause the majority of disease problems for ornamentals in nurseries. Nematodes also may be a major problem in field-grown nursery stock,

but not in soilless container mixes. Bacterial diseases affect some ornamental crops such as crown gall of rose caused by *Agrobacterium tumefaciens*, fireblight caused by *Erwinia amylovora*, and *Pseudomonas* blight of shade trees caused by *P. syringae*. Viruses, although probably more prevalent than commonly realized, do not cause serious disease problems in most woody ornamentals.

Numerous disease problems can be encountered on ornamental plants regardless of the production system used. In many cases, production practices may provide environmental conditions that favor a particular pathogen. Nurseries can grow a crop quite successfully for several years only to be faced with a serious disease problem when cultural practices are altered.

Because there are a large number of plant species grown for the nursery trade, it is not possible to list all the pathogens and diseases affecting these plants. A recent book, *Diseases of Woody Ornamentals and Trees in Nurseries*, lists many important nursery crops and their major diseases along with in-depth control measures.

IPM FOR ORNAMENTAL DISEASES

The development of an IPM program for ornamental diseases requires a knowledge of the source of pathogen inoculum and the effect of environmental factors on inoculum dispersal, infection, host colonization, and disease development. In addition, the role of cultural practices, sanitation, disease resistance, and chemical and biological control must be integrated to develop a successful IPM program for ornamentals.

EXAMPLES OF IPM FOR ORNAMENTALS

Cultural Practices and Sanitation

In the design of production areas for container stock, the layout should allow for adequate drainage during the heaviest thunderstorms to avoid water standing around containers that would favor diseases like *Phytophthora* root rot. Nurseries commonly use crushed

stone as a ground cover to prevent dispersal of *Phytophthora* spp. or other pathogens between containers. Most nurseries recycle irrigation water to meet local requirements that prohibit discharge of nitrate from fertilizer applications into water sheds. Recycled irrigation water is a source of inoculum of *Pythium*, *Phytophthora*, and probably other pathogens that may result in disease development when these pathogens are introduced into containerized plants. Grass strips and vegetation barriers to filter out pathogen inoculum before run off water returns to the retention basin is another example of a cultural practice to reduce the threat of disease. Some nursery operations use chlorination or some other form of water treatment, such as UV light or ozonation, in an attempt to eliminate pathogens from recycled water—an example of sanitation as part of an IPM approach. As with most technologies, however, a chlorination system has to be properly monitored and maintained to achieve satisfactory results.

A new technology in nursery production is pot-in-pot production, wherein the containerized plant is placed in an outer pot that has been buried almost to the soil line. The goal of this production strategy is to produce large plants for the nursery trade and to reduce stress on plants by providing a moderated root zone environment that is not subjected to wide daily fluctuations in temperature and moisture. This cultural practice is an example of an IPM strategy that avoids stressing the plant. Pot-in-pot production minimizes the effects of diseases, such as *Phytophthora* root rot, that result in accentuated symptoms and severity when plants are under stress. This practice also is effective for some foliar pathogens like *Botryosphaeria* spp. and *Phomopsis* spp. that only infect stem tissues when plants are under water stress.

Sanitation is a widely practiced strategy for IPM in ornamental diseases. The goal is to reduce pathogen inoculum in the propagation and production areas. Examples for vegetative propagation of ornamentals include avoiding the use of infested or infected propagation stock, treating cuttings with a disinfectant prior to rooting to eliminate pathogen inoculum, replacing the propagation medium after each crop of cuttings has been rooted, promptly removing diseased cuttings from the propagation house before the pathogen can be dispersed to healthy cuttings, etc. Sanitation strategies for containerized stock include avoiding the reuse of potting mix and containers, pruning out infected shoot tissue, and removing crop debris from the production area that may harbor inoculum. For field-grown stock, similar practices apply. In addition, growers should clean and disinfect machinery that is moved from one block of plants to another to avoid dispersing pathogens. Sanitation is the foundation of any well-designed IPM program for ornamental diseases.

Disease Resistance

An underutilized IPM strategy for ornamental crops is diseases resistance. Traditionally, only a few breeding programs, such as those at the USDA National Arboretum, have tried to incorporate disease resistance into ornamental crops. In most cases, plant pathologists have screened introduced cultivars to find resistance. Increasingly, consumers want disease-free plants for the landscape. Garden centers and retail outlets are beginning to sell plants by advertising disease resistance as a benefit for successful cultivation of a particular cultivar in the landscape. There exists no complete work that contains information on specific resistant cultivars for the various diseases that affect all the different ornamental crops, except the recent book *Diseases of Woody Ornamentals and Trees in Nurseries*. By incorporating disease resistance into an IPM program for ornamentals, all individuals from the propagator, to the producer, to the consumer benefit. Consumer awareness enhanced through effective extension programs and advertising can only enhance the adoption of disease-resistant cultivars for the most popular ornamental crops.

Scouting

As with IPM programs for insect pests, scouting is a very important component of IPM for ornamental diseases. Many large nursery operations have scouts that routinely monitor each block of plants for specific insects and diseases known to be a problem in that particular nursery. Scouting gives the nursery manager a real-time assessment of the disease situation in which appropriate IPM strategies can be applied before remedial measures become hopeless. Early detection of a disease problem can oftentimes result in a satisfactory solution like reducing the amount or timing of irrigation, removing infected tissue before an epidemic starts, or making a pesticide application to prevent further development of disease.

Chemical Control

Fungicides are widely used in IPM programs for ornamental crops. Nematicides are very limited and labeled products have limited effectiveness. Many different bactericides mostly based on copper are available but, again effectiveness is limited. In IPM programs for ornamentals, fungicides are used primarily as preventatives. Therefore, regular calendar-based applications are made routinely for root diseases known to be a problem on a certain crop. For diseases in which pathogen inoculum is not always present, however, like some of the leaf spot diseases, scouting has the potential to reduce fungicide use.

Many new fungicides are becoming available to nurserymen for control of ornamental diseases. These fungicides are based on new chemistry such as the strobilurins that fit into new-risk groups under EPA guidelines providing an opportunity to market them for minor-use crops like ornamentals. Fungicides whether systemic or contact-type chemicals, generally, inhibit the fungal pathogen rather than kill it. Therefore, adequate concentrations of the fungicide must be available on or in host tissues to prevent fungal spore germination and infection. As the concentration of the fungicide drops due to weathering, leaching, and microbial degradation, the fungal propagule can continue its activity unless additional applications are made. Thus, most fungicides are used in a preventative manner on a regular basis when specific diseases problems are known to occur with a particular ornamental crop.

Biological Control

Several biocontrol agents are now available for control of ornamental diseases. Biological control uses beneficial microorganisms, such as *Gliocladium Trichoderma*, *Streptomyces*, *Pseudomonas*, etc., to protect plants from disease. Biocontrol is an ecologically based approach to IPM because this strategy makes use of naturally occurring microorganisms that control diseases. Biocontrol works by inhibiting the germination and growth of the pathogen, killing pathogen cells, creating a competition for nutrients in the root zone or leaf surface, or by inducing the plant to activate natural, host-defense mechanisms. Currently available products are targeted for root diseases caused by *Rhizoctonia*, *Pythium*, *Agrobacterium*, and *Fusarium*. Products are applied as drenches, sprays, dips, or are mixed directly into the potting mix. Widespread adoption of biocontrol for ornamental IPM programs has been hindered by uneven performance of biocontrol agents where widely fluctuating environments are encountered in the nursery. As biocontrol technology improves, this control strategy will become a central component of IPM.

IPM APPROACH

For IPM to be used successfully in the nursery, managers, production leaders, production workers, scouts,

and pesticide applicators must develop a team-based approach so that all have a stake in crop protection. Extension specialists need to interact with the nursery team to help the team understand the various components of the IPM approach and how these strategies can be used effectively in disease management. This requires that the specialist or advisor has a thorough understanding of the nursery at all levels of operation before the specific IPM approach for that nursery is developed. In the future, IPM-based systems may be offered commercially in states or regions where publicly funded assistance is unavailable.

BIBLIOGRAPHY

- Coyier, D.L., Roane, M.K., Eds.; *Compendium of Rhododendron and Azalea Diseases*; APS Press: St. Paul, Minnesota, 1988.
- Hagan, A. Disease resistance. In *Diseases of Woody Ornamentals and Trees in Nurseries*; Jones, R.K., Benson, D.M., Eds.; APS Press: St. Paul, Minnesota, 2001.
- Jeffers, S.N.; Miller, R.W.; Powell, C.C. Fungicides for ornamental crops in the nursery. In *Diseases of Woody Ornamentals and Trees in Nurseries*; Jones, R.K., Benson, D.M., Eds.; APS Press: St. Paul, Minnesota, 2001.
- Jones, R.K., Benson, D.M., Eds.; *Diseases of Woody Ornamentals and Trees in Nurseries*; APS Press: St. Paul, Minnesota, 2001.
- Jones, R.K.; Simone, G.; von Broembsen, S.L.; Dutky, E. Integrated disease management. In *Diseases of Woody Ornamentals and Trees in Nurseries*; Jones, R.K., Benson, D.M., Eds.; APS Press: St. Paul, Minnesota, 2001.
- Moorman, G.W. *Scouting and Controlling Woody Ornamental Diseases in Landscapes and Nurseries*; Penn State College of Agricultural Sciences Publication, (AGRS-62); University Park, Pennsylvania, 1992.
- Sinclair, W.A.; Lyon, H.H.; Johnson, W.T. *Diseases of Trees and Shrubs*; Comstock Publishing Associates, Cornell University Press: Ithaca, New York, 1996.
- von Broembsen, S.L.; MacDonald, J.D.; Pscheidt, J.W. Recycled irrigation water. In *Diseases of Woody Ornamentals and Trees in Nurseries*; Jones, R.K., Benson, D.M., Eds.; APS Press: St. Paul, Minnesota, 2001.
- Williams-Woodward, J.L.; Jones, R.K. Sanitation: plant health from start to finish. In *Diseases of Woody Ornamentals and Trees in Nurseries*; Jones, R.K., Benson, D.M., Eds.; APS Press: St. Paul, Minnesota, 2001.

Papaya Diseases: Ecology and Control

Arevik Poghosyan
Gina Holguin Zehfuss
Macario Bacilio Jimenez

*Programa de Agricultura en Zonas Aridas, CIBNOR, La Paz,
Baja California Sur, Mexico*

INTRODUCTION

Papaya, *Carica papaya* L., is a widely grown fruit in the tropics and subtropics. Besides the fruit, the plant also produces papain, a proteolytic enzyme used in medicine and other industries. Diseases are the most important problems that limit production. Papaya ring spot is a major cause that limits papaya production when compared to other viral diseases, in many countries. The major pathogenic nematodes are reniform and root knot nematodes. Phytoplasma-associated diseases detected recently in different countries are among the most serious ones affecting papaya. The most common bacterial pathogens are *Erwinia* and *Pseudomonas* spp. Fungal pathogens of papaya are numerous.

VIRAL DISEASES

In this section, the diseases with a properly established viral origin, where the causative organisms have been proven to fulfill Koch's postulates are only described. Diseases with inconclusive evidence of viral origin such as papaya droopy necrosis, papaya apical necrosis, and papaya bunchy top disease are included in the section titled "Miscellaneous Diseases." Symptomatically, they seem close to phytoplasma-borne diseases.

Papaya Ring Spot

Papaya ring spot, caused by papaya ring spot virus (PRSV), a potyvirus, is an economically devastating papaya disease worldwide. Only PRSV biotype P infects papaya naturally and probably from the mutation of PRSV-W, a biotype of cucurbits.^[1]

Various symptoms of the disease include stunting, vein clearing, leaf mosaic, mottling, deformation, and stem streaks, and depend on virus strain, plant vigor and size, temperature, and stage of infection. Disease was named so because of the appearance of dark green rings on the fruit skin.^[2,3]

The transmission of PRSV-P takes place through the sap and not through the seed. It is spread by aphids, including *Myzus persicae* and *Aphis gossypii*.^[4]

Control strategies: 1) vector management (insecticides, winged aphid barriers, rouging, and non-host barrier crops); 2) breeding tolerant cultivars; and 3) crossprotection with attenuated virus strain. The latter two strategies have been practiced with varying success.^[2,5]

Although *C. papaya* has no PRSV-resistant gene, the Rainbow and SunUp PRSV-resistant varieties were produced in Hawaii using the virus coat protein gene (CP).^[2,6] Unfortunately, they are not resistant to other strains of the virus, as genetic variation exists among CP genes of PRSV strains from different locations, and also because the virus is highly mutable.^[7] Research is still being carried out in other countries to produce PRSV-resistant transgenic papaya using CP, the viral replicase (RP), and the movement protein (MP) genes.^[6,8]

Papaya Mosaic

Causal agent: papaya mosaic virus (PMV), genus *Potexvirus*. Synonym: papaya (papaw) mild mosaic virus. Reported in U.S.A., Venezuela, Bolivia, Mexico, and Peru. Symptoms: leaf mosaic and stunting. Other facts about the virus: RNA-containing virus, sap-transmissible, vector unknown, and no data on seed transmission. To detect the virus, RT-PCR has been applied successfully.^[9,10]

There are no special control measures against PMV. Infection by PRSV and PMV is often combined, and measures against PRSV might combat PMV. A chemical that induces systemic acquired resistance (SAR) (acibenzolar) in papaya plants might mitigate effects of this viral infection.^[11]

Papaya "Meleira" or "Sticky" Disease

First reported in the 1980s, as the most damaging papaya disease in Brazil. Symptoms: tipburn, and young leaf necrosis owing to latex exudation, more pronounced on green fruits, which darkens as it oxidizes and makes the fruit surface sticky. Affected fruits are malformed, sometimes with blotchy flesh and bad flavor.^[12]

Papaya meleira virus (PmeV), appears to represent a novel group of viruses. PmeV is transmissible through

latex, but not through the sap. Except for papaya, the other hosts of PmeV and their vectors are still not known. The disease is controlled by systematic roguing, from the time of the onset of symptoms.^[12]

NEMATODE-BORNE DISEASES

Nematodes primarily attack roots, affecting plant growth and productivity. They cause stunting, premature wilting, leaf chlorosis, and root malformations. Genera that damage papaya include *Rotylenchus*, *Meloidogyne*, *Helicotylenchus*, *Quinisulcius*, and *Criconemella*, the first two being the most prevalent. They disseminate through cultivation and surface runoff and irrigation.^[8,13]

Reniform Nematodes

The semiendoparasite *Rotylenchus reniformis* is the principal nematode affecting papaya production in North America, South America, the Caribbean Basin, Southern Europe, the Middle East, Asia, Australia, and the Pacific.^[13] Phloem feeding creates giant cells, the centers of high metabolic activity. They also feed in the root cortex, causing mechanical damage and facilitating fungal attack. Female juveniles penetrate the root cortex and become sedentary. Mature females secrete a gelatinous matrix that covers about 60–200 eggs.^[13,14] They can survive at least two years in the absence of a host in dry soil.^[14]

Root Knot Nematodes

They congest root systems, thereby causing swelling and stunting of the plant. The most common ones in papaya are *Meloidogyne incognita*, *M. javanica*, *M. arenaria*, and *M. hapla*. When female larvae feed in the core of the roots, cell number and size increase forming galls or “knots.” Unlike reniform nematodes, the female and associated egg-mass embed in root tissue.^[13]

Control

No known papaya cultivars are resistant to nematodes. Agricultural fields that were formerly used for the cultivation of pineapple and cotton should be avoided. Nematicides that are registered by the EPA are Azadirachtin (from neem *Azadirachta indica* A. Juss), Harpin protein (product of transformed *Escherichia coli* K-12 that induces plant SAR), and DiTera (dried fermentation products of the fungus *Myrothecium verrucaria*).^[8] The main control strategy is to combine nematicides, solarization, agronomic techniques, and biofumigation, as alternatives to highly toxic methyl

bromide.^[15] Biological control methods are developed using antagonists of nematodes *Meloidogyne* spp. and *Tylenchorhynchus cylindricus*: nematophagous fungus *Verticillium chlamidosporium* and endospores of the parasitic bacteria, *Pasteuria*.^[15]

PHYTOPLASMA DISEASES

There are various reports about phytoplasma-associated papaya diseases.^[16] Affected plants are of little commercial value. As phytoplasmas cannot be readily grown in cell-free media, phytoplasma-infection was diagnosed using ultrastructural, serological, and molecular techniques. Using these approaches, three phytoplasma-related diseases were recognized in Australia: dieback (PDB), yellow crinkle (PYC), and mosaic (PM). The most common and devastating of these diseases is the PDB, causing annual losses from 5% to 100%.^[17,18]

Some symptoms are common for all yellow-type diseases in papaya: yellowing and reduction of young leaves, stunting, bending of the stem tip and leafstalks, drying and fall of older leaves, latex flow reduced or absent, and flowers deformed and small. In plants affected by PDB, the death starts from the apical part of the plant, while a nearly total loss of leaves is typical for PYC, with only a few stunted leaves seen in the top. Symptoms of PM include multiple side shoots and mosaic.^[16,17] The vectors of these diseases are unknown, but supposed to be among the insects belonging to the genus *Hemiptera*.^[18]

Restriction fragment length polymorphism analysis of these diseases using 16S rDNA and 16S–23S spacer region revealed that the PYC and PM sequences are identical but distinguishable from PDB. Phylogenetic analysis placed PDB in the taxon *Candidatus* phytoplasma Australiense, whereas PYC and PM are members of *Candidatus* phytoplasma Australasia.^[19]

Plants affected by PDB can be saved if they are cut back to 75 cm when symptoms appear. Only the best two to three new branches should be retained. Regrowth of such plants is free of symptoms. But, in plants affected by PM and PYC, regrowth is usually affected and plants should therefore be removed.^[20]

MISCELLANEOUS DISEASES

Papaya droopy necrosis and a similar disease called papaya apical necrosis have been reported in southern Florida and in Venezuela. The first symptoms of both diseases are drooping and downward cupping of leaves in the upper part of plant. Neither the vector nor an alternative host has been identified. Viral origin for the disease was reported,^[5] but the available data did

not effectively prove this. Diseases are not transmitted mechanically.

Papaya bunchy top disease was observed throughout the Caribbean region. The symptoms of the disease are similar to those of papaya apical and droopy necrosis. The disease was thought to be caused by phytoplasma, but then this was disputed.^[21] Two leafhoppers are known to transmit the bunchy top agent: *Empoasca papayae* and *E. stevensi*. Some papaya cultivars are more tolerant, but immunity is not known. Control: removal of sources of inoculum, by roguing infected trees or topping off the infected plants below the point of latex exudation.^[21]

BACTERIAL DISEASES

The principal bacterial papaya diseases are caused by bacteria belonging to the genera *Pseudomonas* and *Erwinia*. Among the diseases caused by *Pseudomonas*, the most damaging ones are the bacterial leaf spot (*P. carica papaya*) and bacterial wilt (*P. solanacearum*). The diseases caused by *Erwinia* are black rot (*E. cypridiedii*) and decline and mushy canker (*Erwinia* spp.).^[22] Bacterial canker sometimes leads to the destruction of papaya trees. Nineteen strains of *Erwinia* pathogens were recently analyzed by DNA hybridization and were proposed to belong to a novel species, *Erwinia papayae* sp.nov.^[23]

Internal yellowing, caused by *Enterobacter cloacae*, is characterized by yellow discolored flesh, with diffused margins and rotting odor in the fruits. No external symptoms are displayed. Control of *E. cloacae* is currently limited to postharvest hot water quarantine.^[24]

FUNGAL DISEASES

Anthracnose

It is caused by *Colletotrichum gloeosporioides* (Penz.) Penz. and Sacc. in Penz. Anthracnose is a main postharvest papaya disease present on refrigerated fruits that are exported from most tropical and subtropical regions. The fungus attacks primarily the fruit. The first symptoms are round, water-soaked, sunken spots on the ripening fruit. High temperature (28°C) and high relative humidity (97%) favor the pathogen. The fungus is inactive in dry weather, sunlight, and extreme temperatures.^[25,26]

Alternaria Fruit Spot

It is a major fruit disease in dry area orchards. *Alternaria alternate* produces depressed, circular lesions on the fruit surface that blacken because of

the sporulation of the pathogen. Lesions can develop in cold storage (10°C).^[26]

Dry Rot

Common to all commercially grown papaya. *Mycosphaerella* sp. colonizes senescing leaves and petioles, producing fruiting structures (conidia and ascospores) that deposit on the fruit surface during rain, and cause slightly sunken, dry, circular, black lesions that measure up to 4 cm in diameter.^[27]

Cercospora Black Spot

It is caused by *Cercospora papayae* Hansf., and occurs on fruit and leaves. It is common in poorly maintained, nonsprayed papaya fields in wet areas. The small black dots that enlarge to 3 mm in diameter do not develop into fruit rot, but diminish marketability.^[28]

Fusarium Fruit Rot

Fusarium solani (Mart.) Sacc. is the most common disease-causing fungus, and establishes mainly on bruised fruit. Lesions on the fruit surface and stem ends are small and depressed, usually covered by a combination of white mycelia and conidial masses.^[29]

Internal Blight

One or more fungi infect the seed cavity. Most common is the *Cladosporium* sp., but *Fusarium* sp. and *Penicillium* sp. may participate. Fungus grows through the mucilaginous coating, causing it to shrivel, dry, and darken. Infected fruits yellow prematurely.^[30]

Phytophthora Fruit Rot and Root Rot

Phytophthora spp. can cause serious losses during rainy periods by attacking lateral roots and destroying the whole root system. Papaya roots are very susceptible during the first three months after the seedlings emerge. Infected young fruit (*P. palmivora*) show water-soaked lesions that exude milky latex.^[31]

Powdery Mildew

Oidium caricae F. Noackand causes little damage on bearing trees. However, it may severely damage young plants in wet environment, affecting leaves, stems, flower pedicles, and fruits. Greenhouse seedlings are especially susceptible.^[32]

Other Fungal Disease

Soft rot, caused by *Rhizopus stolonifer* (Ehrenb. Fr.) Vuill., and Wet rot (*Phomopsis* sp.) are common during storage and transit. *Stemphylium lycopersici* (Enji) W. Yamamoto produces small, round, dark-brown lesions on fruit; while *Phomopsis* sp. produces wet fruit rot that resembles *Rhizopus* watery soft rot.^[33]

Fungal disease agents recently detected in *C. papaya* are: 1) *Plasmidiophora brassicae* Woron., an obligate biotroph that causes club root disease (tumorous swellings) in the brassicaceae;^[34] 2) *Ustilago mayis* that causes common smut in corn (*Zea mays* L.);^[35] and 3) vascular wilt of highland papayas produced by interaction between *F. oxysporum* and *M. incognita*, the root knot nematode.^[36]

Control of Fungal Diseases

Commercially produced papayas are sprayed with an array of pesticides to control fungi, although some can be phytotoxic. “Reduced impact” chemicals have been introduced recently, including neem oil, Azoxystrobin (active compound strobilurin), and the Harpin protein.

CONCLUSIONS

Numerous diseases limit the production of papaya throughout the world. Among them, PRSV is probably the most devastating. The role of phytoplasma-borne disease and other similar diseases is significant and tends to increase as diagnostic techniques develop.

The introduction of transgenic PRSV-resistant papaya into commercial production has revitalized the industry in some parts of the world. However, public opposition to GM food in Japan, Korea, and Europe jeopardizes the success of the transgenic approach. On top of this, the high mutability of the virus, as well as the genetic variation among PRSV strains from different geographic locations, would require the continuous development along the transgenic lines, for every region. Considering all this, the wisdom of applying a GM approach to disease control is questionable.

A promising method to control some diseases in papaya plants that has been applied successfully to other crops is the inoculation of papaya seeds with beneficial microorganisms that: 1) induce systemic acquired resistance; 2) reduce ethylene levels produced by the plant during pathogen attack; 3) out-compete root-colonizing pathogens by establishing first on the roots; and 4) produce healthy and robust plants.^[37,38]

ACKNOWLEDGMENTS

Gina Holguin participated in this work in memory of the late Mr. Juan Holguin Franco. We thank Taylor Merry for editing the English-language text.

REFERENCES

1. Bateson, M.F.; Lines, R.E.; Reville, P.; Chaleprom, W.; Ha, C.V.; Gibbs, A.J.; Dale, J.L. On the evolution and molecular epidemiology of the potyvirus. *Papaya ringspot virus*. *J. Gen. Virol.* **2002**, *83*, 2575–2585.
2. Gonsalves, D. Papaya ringspot. In *Compendium of Tropical Fruit Diseases*; Ploetz, R.C., Zentmyer, G.A., Nishijima, W.T., Rohrbach, K.G., Ohr, H.D., Eds.; APS Press: Minnesota, U.S.A., 1998; 66–68.
3. Purcifull, D.; Edwardson, J.; Heibert, E. Gonsalves, D. Papaya ringspot virus. In *Descriptions of Plant Viruses, No. 292*; Commonwealth Mycological Institute and Association of Applied Biologists: Kew, England, 1984.
4. Kiritani, K.; Su, H.H. Papaya ring spot, banana bunchy top, and citrus greening in the Asia and Pacific region: occurrence and control strategy. 1998. *Jpn. Agric. Res. Q.* **1999**, *33*; <http://ss.jircas.affrc.go.jp/engpage/jarq/33-1/contents.html> (accessed May 2005).
5. Pernezny, K.; Litz, R.E. Some common diseases of papaya in Florida. *Plant Path. Ext. Rep. No. PP-35*, 1993; <http://www.plantpath.ifas.ufl.edu/takextpub/Fact-Sheets/pp0035.pdf> (accessed May 2005).
6. Chiang, C.H.; Wang, J.J.; Jan, F.J.; Yeh, S.D.; Gonsalves, D. Comparative reactions of recombinant papaya ringspot viruses with chimeric coat protein (CP) genes and wild type viruses on CP-transgenic papaya. *J. Gen. Virol.* **2001**, *82*, 2827–2836.
7. Davis, M.J.; Ying, Z. Development of papaya breeding lines with transgenic resistance to *Papaya ringspot virus*. *Plant Dis.* **2004**, *88*, 352–358.
8. Mosler, M.A.; Nesheim, O.N. *Florida Crop/Pest Management Profile: Papaya, 2002*; Doc.CIR1402. http://edis.ifas.ufl.edu/BODY_PI053htm.
9. Noa Carrazana, J.C.; Silva-Rosales, L. First report of a mexican isolate of papaya mosaic virus in papaya (*Carica papaya*) and pumpkin (*Cucurbita pepo*). *Plant Dis.* **2001**, *85*, 558.
10. Purcifull, D.E.; Heibert, E. Papaya mosaic virus. *AAB Description of Plant Viruses*, 1971; <http://www.dpweb.net>.
11. Mosler, M.A.; Nesheim, O. *Tropical Fruit Pest Management Strategic Plan (PMSP)*; University of Florida, IFAS extension: Florida, 2003; <http://edis.ifas.ufl.edu/>.
12. Maciel-Zambolim, E.; Kunieda-Alonso, S.; Matsuoka, K.; Carvalho, M.G.; Zerbini, F.M. Purification and some properties of papaya meleira virus, a novel virus infecting papaya in Brazil. *Plant Pathol.* **2003**, *52*, 389–394.
13. Holtzmann, O.V.; McSorley, R. Papaya diseases caused by nematodes. In *Compendium of Tropical Fruit Diseases*; Ploetz, R.C., Zentmyer, G.A., Nishijima, W.T., Rohrbach, K.G., Ohr, H.D., Eds.; APS Press: Minnesota, U.S.A., 1998; 68 pp.

14. Ayala, A.; Ramírez, C.T. Host range, distribution and bibliography of the reniform nematode, *Rotylenchulus reniformis*, with special reference to Puerto Rico. *J. Agric. Univ. Puerto Rico* **1964**, *48*, 140–160.
15. Francesco, E. Plant Protection Institute Sect. of Bari (IPP). *Scientific Activity*; <http://www.inaav.ba.cnr.it/inaav2en.htm>. 2005.
16. Poghosyan, A.V.; Lebsky, V.K.; Arce-Montoya, M.; Land, L. Possible phytoplasma disease in papaya (*Carica papaya* L.) from Baja California Sur: diagnosis by scanning electron microscopy. *J. Phytopathol.* **2004**, *152*, 376–380.
17. Gibb, K.S.; Schneider, B.; Padovan, A.C. Differential detection and genetic relatedness of phytoplasmas in papaya. *Plant Pathol.* **1998**, *47*, 325–332.
18. Elder, R.J.; Milne, J.R.; Reid, D.J.; Guthrie, J.N.; Persley, D.M. Temporal incidence of three phytoplasma associated diseases of *Carica papaya* and their potential hemipteran vectors in central and south-east Queensland. *Aust. Plant Pathol.* **2003**, *31* (2), 165–176.
19. White, D.T.; Blackall, L.L.; Scott, P.T.; Walsh, K.B. Phylogenetic position of phytoplasmas associated with dieback, yellow crinkle and mosaic diseases of papaya, and their proposed inclusion in “Candidatus phytoplasma Austarliense” and a new taxon, “Candidatus phytoplasma Australasia.” *Int. J. Syst. Bacteriol.* **1998**, *48* (3), 941–951.
20. Persley, D. *Papaya: Virus and Virus-Like Diseases*; Department of primary industries: Queensland, 1999; www.dpi.qld.gov.au/horticulture/5334.html (accessed 14 August 2003 (HHTT/1. 0 404)).
21. Davis, M.J.; Kramer, J.B.; Ferweda, F.H.; Brunner, B.R. Association of bacterium and not a phytoplasma with papaya bunchy top disease. *Phytopathology* **1996**, *86*, 102–109.
22. Nishijima, K.A. Papaya diseases caused by bacteria. In *Compendium of Tropical Fruit Diseases*; Ploetz, R.C., Zentmyer, G.A., Nishijima, W.T., Rohrbach, K.G., Ohr, H.D., Eds.; APS Press: Minnesota, U.S.A., 1998; 65 pp.
23. Gardan, L.; Christen, R.; Achouak, W.; Prior, P. *Erwinia papayae* sp.nov., a pathogen of papaya (*Carica papaya*). *Int. J. Syst. Evol. Microbiol.* **2004**, *54*, 107–113.
24. Nishijima, K.A. *Enterobacter cloacae*. *Crop Knowledge Master*; University of Hawaii, College of Tropical Agriculture and Human Resources, and Hawaii Department of Agriculture: Hawaii, 1993; <http://www.extento.hawaii.edu/kbase/Crop/Type/e-cloac.htm>.
25. Dickman, M.B.; Alvarez, A.M. Latent infection of papaya caused by *Colletotrichum gloeosporioides*. *Plant Dis.* **1983**, *67*, 748–750.
26. Alvarez, A.M.; Nishijima, W.T. Postharvest diseases of papaya. *Plant Dis.* **1987**, *71*, 681–686.
27. Chau, K.F.; Alvarez, A.M. Role of *Mycosphaerella ascospores* in stem-end rot of papaya fruit. *Phytopathology* **1979**, *69*, 500–503.
28. Hine, R.B.; Holtzmann, O.V.; Raabe, R.D. Diseases of papayas (*Carica papaya* L.) in Hawaii. *Univ. Hawaii Agric. Exp. Stn. Bull.* **1965**, *136*.
29. Saxena, R.M.; Sharma, K.D. Deterioration of papaya fruits by fungi. *Agric. Sci. Dig.* **1981**, *1*, 140–142.
30. Zee, F.T.; Nishina, M.S.; Chan, H.T.; Nishijima, K.A. Blossom end defects and fruit fly infestation in papayas following hot water quarantine treatment. *HortScience* **1989**, *24*, 323–325.
31. Hunter, J.E.; Kunimoto, R.K. Dispersal of *Phytophthora palmivora* sporangia by wind-blown rain. *Phytopathology* **1974**, *64*, 202–206.
32. Raabe, R.D. Susceptibility of senescent leaves fruiting papaya plants to powdery mildew. *Plant Dis. Rep.* **1966**, *50*, 519.
33. Nishijima, W.T. Rhizopus soft rot. In *Compendium of Tropical Fruit Diseases*; Ploetz, R.C., Zentmyer, G.A., Nishijima, W.T., Rohrbach, K.G., Ohr, H.D., Eds.; APS Press: Minnesota, U.S.A., 1998; 63 pp.
34. Ludwig-Müller, J.; Bennett, R.N.; Kiddle, G.; Ihmig, S.; Ruppel, M.; Hilgenberg, W. The host range of *Plasmodiophora brassicae* and its relationship to endogenous glucosinolate content. *New Phytol.* **1999**, *141*, 443–458.
35. León Ramírez, C.G.; Cabrera-Ponce, J.L.; Martínez-Espinosa, A.D.; Herrera-Estrella, L.; Méndez, L.; Reynaga-Peña, C.G.; Ruiz-Herrera, J. Infection of alternative host plant species by *Ustilago maydis*. *New Phytol.* **2004**, *164*, 337–346.
36. Ochoa, J.B.; Fonseca, G.G.; Williams, R.N.; Ellis, M.A. Interactions between *Fusarium oxysporum* and *Meloidogyne incognita* causing vascular wilt of highland papayas in Ecuador. *Fitopatologia* **2003**, *38*, 170–175.
37. Glick, B.R.; Patten, C.L.; Holguin, G.; Penrose, D.M. *Biochemical and Genetic Mechanisms Used by Plant Growth Promoting Bacteria*; Imperial College Press: London, U.K., 1999.
38. Van Loon, L.C.; Bakker, P.A.H.M.; Pieterse, C.M.J. Systemic resistance induced by rhizosphere bacteria. *AR Phytopathol.* **1998**, *36*, 453–483.

Papaya Insects: Ecology and Control

Alberto Pantoja

Agricultural Research Service, Subarctic Agricultural Research Unit,
United States Department of Agriculture, Fairbanks, Alaska, U.S.A.

Jorge E. Peña

Tropical Research and Education Center, University of Florida,
Homestead, Florida, U.S.A.

INTRODUCTION

Papaya, *Carica papaya* L., originated in tropical America, and it is currently grown in all tropical and in many subtropical regions of the world. Papaya is mainly cultivated for its edible fruit, but medical and industrial uses have been documented.^[1,2] There are 134 species of arthropods that affect papaya.^[3] Most of the species belong to the Hexapoda, while 12 belong to the Arachnida (Table 1).

Twenty-six species are fruit flies in the family Tephritidae. Eighty-seven species can potentially attack or damage the fruit but are mainly associated with the foliage or the trunk. One species is a seed borer. Five species affect the flowers, and three species are root feeders. At least 12 species are known vectors of important papaya diseases. In different papaya growing areas, fruit flies (Diptera: Tephritidae), leafhoppers (Homoptera: Cicadellidae), Aphids (Homoptera: Aphididae), mites (Acarina), and mealybugs and scale insects (Homoptera: Coccidae, Conchaspididae, Pseudococcidae, Diaspididae, Asterolecaniidae, and Margarodidae) are considered key pests requiring frequent pesticide applications. Fruit flies are the most important papaya pests either because of their direct effect on the fruit or for quarantine-related issues. Aphids and leafhoppers are key pests because of their vector capacity and mealybugs and scales for quarantine-related issues.

PAPAYA INSECTS

Papaya fruit flies (Diptera: Tephritidae) are the only group of insects that actually penetrate the pulp or seeds. Twenty-six species from seven genera, *Anastrepha*, *Bactrocera*, *Ceratitis*, *Dacus*, *Euphranta*, *Myoleja*, and *Toxotrypana*, attack papaya fruits (Table 1). *Toxotrypana curvicauda* is the most important fruit fly attacking papaya in the Americas and Caribbean Basin, whereas *Bactrocera papayae* is one of the most threatening pests to papaya in Australia.

With the exception of *T. curvicauda*, eggs of fruit flies are regularly laid below the skin of the ripening

fruit and typically hatch in one to four days. *T. curvicauda* lays eggs in the fruit cavity. Medflies [*Ceratitis capitata* (Wiedemann)] and melon flies [*Bactrocera cucurbitae* (Coquillett)] lay 10 to 15 eggs per day, whereas oriental fruit flies [*Bactrocera dorsalis* (Hendel)] lay 130 eggs per day usually in groups of 10, but sometimes as many as 100 or more. Larvae will feed for one to four weeks, depending on temperature, and drop from the fruit to pupariate in the soil under the papaya plant. Adults emerge in one to two weeks.

The Asian papaya fruit fly *B. papayae*, a polyphagous species, and the papaya fruit fly *T. curvicauda*, a new world stenophagous species, are considered to be the most damaging insect pest of papayas.^[4] The female *T. curvicauda* lay about 10 eggs per fruit, predominantly in green fruits.

Sampling and Monitoring

Most reports on the papaya fruit fly *T. curvicauda* are from the U.S.A. and Mexico.^[3] Studies during the mid 1980s and early 1990s concentrated on adult behavior and the male sex pheromone, oviposition and feeding behavior on papaya seeds, and daily activity patterns and within-field distribution of the papaya fruit fly. The use of pheromone traps for *T. curvicauda* has been studied by several researchers.^[5] In Hawaii and Australia, fruit flies are monitored using traps baited with male lures.

Control

Several methods have been reported for papaya fruit fly control, including cultural and chemical control measures. Traditional measures involve insecticides and non-protein toxic baits.^[5,6] Other control measures include destruction of infested fruits, removal of wild hosts, and heat treatments.^[7]

Population suppression in papaya fields can be achieved by several methods. Sanitation is one of the methods. In Hawaii, sanitation is usually insufficient

Table 1 Arthropods associated to papaya

| Taxonomic group | Distribution | Part of plant affected |
|--|--|------------------------|
| Hemiptera | | |
| <i>Coreidae</i> | | |
| <i>Amblypelta cocophaga</i> China | PI, AU | FR |
| <i>Amblypelta costalis szentivanyi</i> Van Duzee | PI, AU | FR |
| <i>Amblypelta gallegonis</i> Lever | PI, AU | FR |
| <i>Amblypelta lutescens papuensis</i> (Distant) | PI, AU | FR |
| <i>Amblypelta theobromae</i> Brown | PI, AU | FR |
| <i>Brachylybas variegates</i> Le Guillou | PI | FR |
| <i>Miridae</i> | | |
| <i>Fulvius angustatus</i> Usinger | PI | FR |
| <i>Pentatomidae</i> | | |
| <i>Nezara viridula</i> (L.) | PI, WI | FR |
| <i>Tingidae</i> | | |
| <i>Corythucha gossypii</i> (F.) | WI | FL |
| Homoptera | | |
| <i>Coccidae</i> | | |
| <i>Coccus discrepans</i> (Green) | AS, ME | FO, FR, TR |
| <i>Coccus hesperidum hesperidum</i> L. | AF, AS, AU, CA, EU, ME, NA, P., SA, WI | FO, FR, TR |
| <i>Coccus longulus</i> (Douglas) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Drepanococcus chiton</i> (Green) | AS, PI | FO, FR, TR |
| <i>Eucalymnatus tessellatus</i> (Signoret) | AF, AS, AU, EU, NA, PI, SA, WI | FO, FR, TR |
| <i>Milviscutulus mangiferae</i> (Green) | AF, AS, CA, EU, NA, PI SA, WI | FO, FR, TR |
| <i>Parasaissetia nigra</i> (Nietner) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Philephedra tuberculosa</i> Nakahara & Gill | CA, NA, SA | FO, FR, TR |
| <i>Protopulvinaria pyriiformis</i> (Cockerell) | AF, AS, CA, EU, NA, PI, SA, WI | FO, FR, TR |
| <i>Saissetia oleae oleae</i> (Olivier) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Saissetia coffeae</i> (Walker) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Conchaspidae</i> | | |
| <i>Conchaspis angraeci</i> Cockerell | AF, AS, AU, CA, EU, NA, PI, SA, WI | FO, FR, TR |
| <i>Pseudococcidae</i> | | |
| <i>Dysmicoccus nesophilus</i> Williams & Watson | PI | FO, FR, TR |
| <i>Ferrisia virgata</i> (Cockerell) | AF, AS, AU | FO, FR, TR |
| <i>Nipaecoccus viridis</i> (Newstead) | AF, AS, AU | FO, FR, TR |
| <i>Paracoccus marginatus</i> Williams & | CA, SA, WI | FO, FR, TR |
| Granara de Willink | | |
| <i>Planococcus citri</i> (Risso) | PI | FO, FR, TR |
| <i>Pseudococcus jackbeardsleyi</i> Gimpel & Miller | AS, CA, NA, PI, SA, WI | FO, FR, TR |
| <i>Pseudococcus longispinus</i> (Targioni Tozzetti) | AF, AS, AU | FO, FR, TR |
| <i>Pseudococcus viburni</i> (Signoret) | AF, AS, AU, PI | FO, FR, TR |
| <i>Diaspididae</i> | | |
| <i>Aonidiella aurantii</i> (Maskell) | PI | FO, FR, TR |
| <i>Aonidiella comperei</i> Mckenzie | PI | FO, FR, TR |
| <i>Aonidiella inornata</i> Mckenzie | PI | FO, FR, TR |
| <i>Aonidiella orientalis</i> (Newstead) | AF, ME, PI | FO, FR, TR |
| <i>Aspidiotus destructor</i> Signoret | AS, PI | FO, FR, TR |
| <i>Aspidiotus excisus</i> Green | PI | FO, FR, TR |
| <i>Aspidiotus macfarlanei</i> Williams & Watson | PI | FO, FR, TR |
| <i>Chrysomphalus dictyospermi</i> (Morgan) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Howardia biclavis</i> (Comstock) | PI | FO, FR, TR |
| <i>Morganella longispina</i> (Morgan) | PI | FO, FR, TR |
| <i>Pseudaonidia trilobitiformis</i> (Green) | PI | FO, FR, TR |
| <i>Pseudaulacaspis cockerelli</i> (Cooley) | PI | FO, FR, TR |
| <i>Pseudaulacaspis pentagona</i> (Targioni-Tozzetti) | WI, PI | FO, FR, TR |
| <i>Pseudoparlatoria ostreata</i> Cockerell | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Asterolecaniidae</i> | | |
| <i>Asterolecanium pustulans</i> (Cockerell) | AS, PI | FO, FR, TR |
| <i>Margarodidae</i> | | |
| <i>Icerya aegyptiaca</i> (Douglas) | PI | FO, FR, TR |
| <i>Icerya purchasi</i> Maskell | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FO, FR, TR |
| <i>Icerya seychellarum</i> (Westwood) | AF, AS, PI | FO, FR, TR |

(Continued)

Table 1 Arthropods associated to papaya (*Continued*)

| Taxonomic group | Distribution | Part of plant affected |
|--|--|------------------------|
| <i>Steatococcus samaraius</i> Morrison | PI | FO, FR, TR |
| <i>Cicadellidae</i> | | |
| <i>Empoasca papayae</i> Oman | SA, WI | FO, VE |
| <i>Empoasca canavalia</i> Long | WI | FO |
| <i>Empoasca dilatata</i> DeLong & Davidson | WI | FO |
| <i>Empoasca fabalis</i> Harris | WI | FO, VE |
| <i>Empoasca insularis</i> Oman | WI | FO, VE |
| <i>Empoasca stevensi</i> Young | WI, PI, NA | FO, VE |
| <i>Empoasca solana</i> DeLong | PI, WI | FO |
| <i>Poeciloscarta laticeps</i> Metcalf & Bruner | WI | FO |
| <i>Sanctanus fasciatus</i> (Osborn) | WI | FO |
| <i>Cixiidae</i> | | |
| <i>Oliarus complectus</i> Ball | WI | FO |
| <i>Derbidae</i> | | |
| <i>Omoligna puertana</i> Caldwell | WI | FO |
| <i>Aleyrodidae</i> | | |
| <i>Trialetrodes variabilis</i> (Quaintance) | WI | FO |
| <i>Aleurocanthus woglumi</i> Ashby | AF, AS, AU, CA, EU, ME, NA, P., SA, WI | FO |
| <i>Aleurodicus destructor</i> (Mackie) | PI | FO |
| <i>Aleurodicus dispersus</i> Russell | CA, EU, NA, PI, SA, WI | FO |
| <i>Tetraletrodes acaciae</i> (Quaintance) | CA, NA, PI., SA, WI | FO |
| <i>Aphididae</i> | | |
| <i>Aphis coreopsidis</i> (Thomas) | WI | FO |
| <i>Aphis craccivora</i> Koch | PI | FO, VE |
| <i>Aphis gossypii</i> Glover | PI, WI | FO, VE |
| <i>Aphis middletonii</i> (Thomas) | PI | FO |
| <i>Aphis nerii</i> Boyer de Fonscolombe | WI | FO, VE |
| <i>Aphis spiraeola</i> Patch | WI | FO, VE |
| <i>Macrosiphum euphorbiae</i> (Thomas) | PI | FO, VE |
| <i>Myzus persicae</i> (Sulzer) | PI, WI | FO, VE |
| <i>Rhopalosiphum maidis</i> (Fitch) | PI | FO, VE |
| <i>Toxoptera aurantii</i> (Boyer de Fonscolombe) | WI | FO |
| Thysanoptera | | |
| <i>Thripidae</i> | | |
| <i>Selenothrips rubrocinctus</i> (Giard) | PI | FO, FL, FR |
| <i>Thrips tabaci</i> Lindeman | PI | FO, FL, FR |
| <i>Frankliniella occidentalis</i> (Pergande) | PI | FO, FL, FR, VE |
| <i>Frankliniella fusca</i> (Hinds) | PI | FO, FL, FR, VE |
| Coleoptera | | |
| <i>Curculionidae</i> | | |
| <i>Acicnemis crassiusculus</i> Fairmaire | PI | FO |
| <i>Diaprepes abbreviatus</i> (L.) | NA, WI | FO, RO |
| <i>Rhabdoscelus obscurus</i> (Boisduval) | PI | FO |
| <i>Metamasius hemipterus</i> (Linnaeus) | CA, NA, SA, WI | RO |
| <i>Anthribidae</i> | | |
| <i>Araecerus vieillardii</i> Montr. | PI | TR |
| <i>Nitidulidae</i> | | |
| <i>Carpophilus maculatus</i> Murray | PI | FR |
| <i>Scarabaeidae</i> | | |
| <i>Protaetia orientalis</i> (Gory & Percheron) | PI | RO |
| Diptera | | |
| <i>Tephritidae</i> | | |
| <i>Anastrepha ludens</i> (Loew) | CA, NA | FR |
| <i>Anastrepha suspensa</i> (Loew) | NA, WI | FR |
| <i>Bactrocera bryoniae</i> (Tyron) | PI | FR |
| <i>Bactrocera cucumis</i> (French) | AU | FR |
| <i>Bactrocera cucurbitae</i> (Coquillett) | AF, AS, ME, PI | FR |
| <i>Bactrocera dorsalis</i> (Hendel) | AS, PI | FR |

(Continued)

Table 1 Arthropods associated to papaya (*Continued*)

| Taxonomic group | Distribution | Part of plant affected |
|---|--|------------------------|
| <i>Bactrocera diversa</i> (Coquillett) | AS, | FR |
| <i>Bactrocera facialis</i> (Coquillett) | PI | FR |
| <i>Bactrocera frauenfeldi</i> (Schiner) | AU, PI | FR |
| <i>Bactrocera jarvisi</i> (Tryon) | AU | FR |
| <i>Bactrocera kirki</i> (Froggatt) | PI | FR |
| <i>Bactrocera melanota</i> (Coquillett) | PI | FR |
| <i>Bactrocera musae</i> (Tryon) | PI | FR |
| <i>Bactrocera neohumeralis</i> (Hardy) | PI | FR |
| <i>Bactrocera passiflorae</i> (Froggatt) | PI | FR |
| <i>Bactrocera trilineola</i> Drew | PI | FR |
| <i>Bactrocera tryoni</i> (Froggatt) | AU, PI | FR |
| <i>Bactrocera xanthodes</i> (Broun) | PI | FR |
| <i>Bactrocera zonata</i> (Saunders) | AS, AU, PI | FR |
| <i>Ceratitis capitata</i> (Wiedemann) | AF, AS, AU, CA, EU, ME, NA, PI, SA, WI | FR |
| <i>Ceratitis catoirii</i> Guérin-Méneville | AF | FR |
| <i>Ceratitis rosa</i> Karsch | AF, AS | FR |
| <i>Dacus bivittatus</i> (Bigot) | AF | ? |
| <i>Euphranta lemniscata</i> (Enderlein) | AS, PI | FR |
| <i>Myoleja nigroscutellata</i> (Hering) | AS, ME | NR |
| <i>Toxotrypana curvicauda</i> Gerstaecker | CA, NA, SA, WI | FR, SE |
| Lepidoptera | | |
| Noctuidae | | |
| <i>Epitomiptera orneodalis</i> (Guenee) | WI | FO |
| <i>Agrotis ipsilon</i> Hufnagel | PI, WI | FO |
| <i>Eudocima fullonia</i> (Clerck) | PI | FO, FR |
| <i>Tiracola plagiata</i> (Walker) | PI | FO |
| Sphingidae | | |
| <i>Erynnis alope</i> (Drury) | SA, WI | FO |
| <i>Erynnis ello</i> (L.) | CA, NA, SA, WI | FO |
| <i>Erynnis lassauxi merianae</i> Grote | WI | FO |
| Phycitidae | | |
| <i>Davara caricae</i> Dyar | WI | FO, FR, TR |
| Tortricidae | | |
| <i>Amorbia emigratella</i> Busck | CA, NA | FO |
| <i>Adoxophyes fasciculana</i> Walker | PI | FO, FR |
| <i>Decadarchis minuscula</i> Wals | PI | FO |
| Acarina | | |
| Eryophidae | | |
| <i>Calacarus citrifolli</i> Keifer | WI | FO |
| <i>Calacarus brionese</i> Keifer | WI | FO, FR |
| Tarsonemidae | | |
| <i>Polyphagotarsonemus latus</i> (Banks) | AU, AS, AF, NA, SA, PI, WI | FO, FR |
| Tydeidae | | |
| <i>Tydeus</i> spp. | WI | FO, FR |
| Tetranychidae | | |
| <i>Eutetranychus banksi</i> (McGregor) | PI | FO, FR |
| <i>Tetranychus cinnabarinus</i> (Boisduval) | AF, ME, PI, WI | FO, FR, VE |
| <i>Tetranychus urticae</i> Koch | SA, WI, NA | FO, FR |
| <i>Tetranychus tumidus</i> (Banks) | WI | FO, FR |
| <i>Tetranychus truncatus</i> Ehara | PI | FO, FR |
| Tenuipalpidae | | |
| <i>Brevipalpus phoenicis</i> (Geijskes) | SA, PI, WI | FO, FR |
| Tuckerellidae | | |
| <i>Tuckerella ornata</i> (Tucker) | PI, WI | FO |
| <i>Tuckerella pavoniformis</i> Ewing | CA, SA, PI, WI | FO |

AF = Africa, AS = Asia, AU = Australia, CA = Central America, EU = Europe, ME = Middle East, NA = North America, PI = Pacific Islands, SA = South America, WI = West Indies and Caribbean; FR = fruits, FO = foliage, FL = flowers, RO = roots, SE = seed, TR = trunk, VE = vector, NR = not recorded, ? = doubtful host.

(Adapted from Ref.^[3].)

by itself because fruit flies are abundant on alternate host plants and can fly in from outside areas. Insecticide protection is possible using cover or bait sprays. Malathion is the most commonly used insecticide, but the microbe-derived toxin spinosad is becoming a widely accepted alternative to malathion. Biological control has been tried with fruit flies with little success, but the potential of inundative parasitoid releases, alone or with bait sprays, is being studied. *Doryctobracon toxotrypanae* (Marsh) attacking *T. curvicauda* has been reported in Costa Rica.^[3] The most effective parasitoid enemy of medfly and oriental fruit fly in Hawaii is *Fopius arisanus*.^[8] In Thailand, *Diachasmimorpha longicaudata* is responsible for 42% reduction of *B. papayae* densities.^[9] Male annihilation, using attraction of males to insecticide-laced lures, and sterile insect techniques, using releases of large numbers of sterile flies to disrupt reproduction, have been used elsewhere to eradicate fruit flies, but these tactics are not presently considered feasible in Hawaii. Harvesting early is an effective means to avoid fruit fly damage. Differences in varietal susceptibility to *T. curvicauda* have been documented for the Hawaiian and Cera varieties.

Arthropods affecting the foliage and trunk of papaya include scales (Homoptera: Coccidae, Conchaspidae, Pseudococcidae, Diaspididae, Asterolecaniidae, and Margarodidae), aphids (Homoptera: Aphididae), leafhoppers (Homoptera: Cicadellidae), hornworms (Lepidoptera: Sphingidae), and mites (Acarina) (Table 1). We will limit our discussion to mites, leafhoppers, aphids, and mealybugs. Nine cicadellid species from three genera (*Empoasca*, *Poeciloscarta*, and *Sanctanus*) can affect papaya. Leafhoppers cause two types of damage: direct feeding and secondary damage as vectors. Symptoms of leafhopper feeding include tip burn, wrinkling and cupping of the leaves, burning of leaf margins in large trees, and stunting of smaller plants. Leafhoppers are more important for their vectoring ability than for the mechanical damage.

In the Caribbean Region, papaya production is severely limited by papaya bunchy top disease, transmitted by *E. papayae*, *E. stevensi*, and *E. insularis*.

Aphids (Aphididae) do not colonize papaya plants, but several species (Table 1) can be found on papaya plants or collected on water pan traps in papaya fields. Aphids are considered a serious threat to papaya production because of their ability to transmit diseases, in particular papaya rin spot virus (PRSV) and the papaya mosaic virus. Several aphid species are capable of transmitting PRSV to papayas in Hawaii, Mexico, and Puerto Rico. Recently, through genetic transformation with PRSV-resistant variety of papaya, "Rainbow," has been developed.^[10]

Refined oil sprays have been suggested as physical barriers for viral transmission. However, inconsistent

results and high control costs prevent wide adoption of this technology. Integrated Crop Management strategies for papaya aphids have been developed in Mexico and the Philippines to manage the viral disease and the vectors. Barrier crops have been proposed as a way to interfere on aphid landing and searching behavior. The use of companion crops such as sorrel (*Hibiscus sabdariffa* L.) may reduce virus incidence by interfering with host finding. Intercropping barriers of corn or sorghum are used as intermediate landing crops in the Philippines. Protecting the seedlings under polypropylene or antiaphid covers are recommended to reduce rapid field infestations.

P. marginatus, is a pest of papaya, cassava, *Hibiscus*, eggplant, avocado, annona, and sweet potato.^[11] The insect has been reported from papaya in Mexico the Caribbean islands of Antigua, Belize, British Virgin Islands, the Dominican Republic, Guatemala, Haiti, Nevis, St. Kitts, Puerto Rico, the US Virgin Islands, Costa Rica, and from the continental U.S.A. (Florida) since 1998. The insects feed on leaves, stems, fruits, and even on seedlings. Mealybugs cause deformation, wrinkling and rolling of the leaf edges, and early leaf drop. Attack to unripe fruits causes sap running and blemishes, a source of fruit downgrading.

Biological control appears to be the main factor keeping the species under control in Mexico, where the most important natural enemies are *Anagyrus* sp., *Acerophagus* sp., and *Apoanagyrus* sp. Common predators are *Chrysopa* sp. and *Chilocorus cacti* L. but usually are found in low densities. Owing to its potential pest status in the Caribbean region, a classical biological control program against *P. marginatus* was initiated involving introduction of parasites from Mexico into the Bahamas and Florida, U.S.A.

Twelve species of mites in seven genera affect papaya (Table 1). Mites are probably the most persistent arthropod pests of papaya. The lack of basic information on mite biology and ecology on papaya has prevented the development of effective management practices. Naturally occurring predators can suppress mite populations after pesticides are removed from the system. However, most producers apply insecticides on a calendar basis, disrupting the natural pest balance. In Hawaii, during early spring, when natural enemies are low, and plants are susceptible, mite populations can reach densities that trigger the use of disruptive acaricides and a pesticide treadmill begins for the rest of the season. In Hawaii, the carmine mite, *Tetranychus cinnabarinus* (a key pest), the red and black flat mite, *Brevipalpus phoenicis* (an occasional pest), and the papaya leaf edge roller mite, *Calacarus brionese*, are common. In other tropical areas, the broad mite *Polyphagotarsonemus latus* causes injury that is sometimes confused with symptoms of PRSV and bunchy top.

CONCLUSIONS

A clear understanding of the pest biology, behavior, population dynamics, and pest status is the foundation for the development of Integrated Pest Management (IPM) strategies. Unfortunately, in spite of the economic importance and wide geographical distribution of the crop, papaya pest control, with the exception of fruit flies, has been poorly studied. Currently, no IPM program is available, even for an insect complex like fruit flies where abundant information on behavioral responses to pheromone and host finding, trapping systems, habitat manipulation, orchard design, and sanitation practices for fruit flies management is available. Current and recent developments in the integration of sampling and the use of food attractants and insecticides have allowed a reduction in the use of broad spectrum pesticides; however, farmers rely heavily on insecticide use and postharvest treatments to manage fruit flies. Research is needed on biologically and culturally based practices to manage indirect pests and to integrate all available tactics for insects damaging the fruit.

Aphids and leafhoppers are important pests of papaya in the Americas and the Caribbean mainly because of their vectoring capacity. Factors affecting host finding and colonization by aphids and leafhoppers need to be studied and integrated to existing cultural practices for other pests, mainly fruit flies. Virus-resistant papaya varieties are available, but the stability of resistance is unknown and varieties might not be available to small farm settings in Latin America and Africa. Further work is needed on aphid sampling, host finding, colonization, and insect-pathogen relationships. Papaya bunchy top is still a limiting factor for papaya production in the Caribbean, but little work has been conducted on the vector biology, sampling, natural enemies, and the pathogen-insect-plant relationship. Only poor to modest relationships have been shown between aphids and leafhopper vectors and the number of affected plants in a field. It is therefore unclear how chemical control of adults will reduce damage.

Biological and cultural control tactics on papaya-based systems need further attention. Culturally based practices can provide a first line of defence against secondary pests, and such practices are available for other crops and in countries producing papaya. Research and extension protocols should emphasize integrating cultural and biologically based practices to develop IPM and integrated crop management programs.

ACKNOWLEDGMENTS

Recognition is extended to Dennis Fielding, Loretta Winton, and Todd Adams, USDA, ARS, Fairbanks Alaska, for critical review of the manuscript.

REFERENCES

1. Morton, J.F. *Mayor Medicinal Plants*; C.C. Thomas Publishing: Springfield, Illinois, 1977.
2. Duke, J.A. *Handbook of Energy Crops*. Available at: http://www.hort.purdue.edu/newcrop/duke_energy/dukeindex.html (accessed January 2005).
3. Pantoja, A.; Follet, P.A.; Villanueva, J.A. Papaya pests. In *Tropical Fruit Pests and Pollinators*; Peña, J.E., Sharp, J.L., Wysolosky, M., Eds.; CABI Publishing: Wallingford, U.K., 2002; 131–156.
4. Heath, R.R.; Epsky, N.D.; Jiménez, A.; Dueben, B.D.; Landolt, P.J.; Meyer, W.L.; Aluja, M.; Rizzo, J.; Camino, M.; Jerónimo, F.; Baranowski, R.M. Improved pheromone-based trapping systems to monitor *Toxotrypana curvicauda* (Diptera: Tephritidae). *Florida Entomol.* **1996**, *79*, 37–47.
5. Landolt, P.J. Behavior of the papaya fruit fly *Toxotrypana curvicauda* Gerstaecker (Diptera: Tephritidae) in relation to its host plant, *Carica papaya* L. *Folia Entomol. Mexicana* **1984**, *61*, 215–224.
6. Sharp, J.L.; Landolt, P.J. Gustatory and olfactory behavior of the papaya fruit *Toxotrypana curvicauda* Gerstaecker (Diptera: Tephritidae) in the laboratory with notes on longevity. *J. Georgia Entomol. Soc.* **1984**, *19*, 176–182.
7. Follett, P.A.; Gabbard, Z. Efficacy of the papaya vapor heat quarantine treatment against white peach scale in Hawaii. *Horticult. Technol.* **1999**, *9*, 506.
8. Vargas, R.I.; Stark, J.D.; Uchida, G.K.; Purcell, M. Opiine parasitoids (Hymenoptera: Braconidae) of oriental fruit fly (Diptera: Tephritidae) on Kauai island, Hawaii: island wide relative abundance and parasitism rates in wild and orchard guava habitats. *Environ. Entomol.* **1993**, *22*, 246–253.
9. Petcharat, J. A small field trial on the release of *Dia-chasmimorpha longicaudata* Ashmead (hymenoptera: Braconidae) in an attempt to control the fruit fly *Bactrocera papayae* Drew and hancock (Diptera: Tephritidae) population. *Khon Kaen Agric.* **1997**, *25*, 62–65.
10. Fitch, M.M.M.; Manshardt, R.M.; Gonsalves, D.; Slightom, J.L.; Sanford, J.C. Virus resistant papaya plants derived from tissues bombarded with coat protein gene of papaya ringspot virus. *Biotechnology* **1992**, *10*, 1466–1472.
11. Peña, J.E.; Sharp, J.L.; Wysosoky, M. *Tropical Fruit Pests and Pollinators*; CABI Publishing: Wallingford, U.K., 2002.

Parasites on *Oulema (Lema) lichenis* Voet, 1826

Ján Gallo

Department of Plant Protection, Slovak Agricultural University, Nitra, Slovak Republic

INTRODUCTION

The determination of criteria of pest harmfulness seems to be the first stage of the ecological approach to solving problems of plant control. Natural enemies of pests (including parasites) have an important role in this approach and, under certain conditions, are able to maintain the pest population on an economically meaningless level of harmfulness for a relatively long time. In this article, I present a review of parasites that thrive on pupas or larvae of *Oulema lichenis*, and outline some possibilities of their application in biological pest control.

There live in Europe two species of *Lema* beetles in all cereal species, representative of the genus *Oulema* (Gozis, 1886): *O. (Lema) lichenis* (Voet, 1826 = Linnaeus, 1758), syn. *O. gallaeciana* (Heyden, 1870); and *Lema melanopus* (Linnaeus, 1758). For some years, feeding by their imagines and larvae reached an intensity such that leaves were fully destroyed. There is clearly an increase in the occurrence of *Lema*, not only in our country but also in neighboring countries (e.g., Czech Republic, Poland, Hungary, Germany, and Bulgaria).^[1] An intensive application of pesticides did not prevent the overspreading of *Lema* and consequent damages. Therefore research has been aimed at detecting their natural enemies so that the most effective parasite species in biological control can be directed against these pests.

ECONOMIC IMPORTANCE OF LEMA BEETLES

During calamitous years, the yields of cereal crops in areas attacked by *Lema* can be reduced by as much as 25%. The destruction of the winter wheat leaf area by 10% reduces the yield of grain by 3.5%. The loss of the leaf area amounting to 13–15% leads to a 14% crop reduction. Detrimental occurrence of larvae is a reality when more than eight beetles per 1 m² are found. Harmfulness depends on the growth phase of cereal species. Early voracity increases loss and, according to Balachowsky,^[2] the highest harmfulness occurs in areas of extensive grain production in the Central and East Europe because yield loss here is usually 50–100%. The damage depends on the grain variety as well as on the intensity of voracity. Spring cereals are more sensitive than winter ones. The

destruction of the winter wheat leaf area by 10% reduces crop yield by 3.5%. A 13–15% destruction of the leaf area leads to a crop loss of 14%, and a 90% destruction reduces crop yield by 23%. As to maize, a 40–50% destruction of its leaf area caused by the voracity of *L. melanopus* did not manifest in crop reduction. For the mass occurrence of both species, the temperature in the two last decades of May (the time of ovum laying) is decisive. If the temperature in this period exceeds the mean decade temperature measured for several years, then a strong occurrence of larvae is highly probable. The higher harmfulness of *Lema* can be expected in the year that follows a year with a mild winter, which creates favorable preconditions for the hibernation of beetles and fertility of females. The threshold of harmfulness in various countries is judged very differently.^[3] The regulation of *Lema*, according to some authors,^[4] is affected by the occurrence of predatory insect and parasites.

LEMA AS HOST OF PARASITES

Besides ecological factors, some authors also analyzed the importance of predacious and parasitic insects on the regulation of the occurrence of *Lema* and, sporadically, parasites from the *Diptera* series were reared.^[4] According to Haeselbarth,^[5] the most abundant parasite on the *Lema* pupas is *Necremnus leucarthros* (Eulophidae). According to Šedivý,^[1] 12 species of hymenopterous parasites were reared in pupas of *O. gallaeciana* Heyd. In addition, six species from the family Ichneumonidae can be named: *Bathytrix maculatus*, *Gelis instabilis*, *Lemophagus curtus*, *Itopectis alternans*, *I. maculator*, and *Scambus annulatus*. Other species comes from the family Pteromalidae: *Pteromalus chrysos*, *P. semotus*, *P. vibulens*, and *Trichomalopsis microptera*. From the family Eulophidae, only the species *N. leucarthros* can be named. Other authors give analogical reviews of parasites.^[5,6] Dysart, Maltby, and Brunson^[7] state the following predators (*Nabis feroides*, *Coccinella septempunctata*, and *Polystes* spp.), parasites (*Anaphes flavipes*, *Tersilochus carinifer*, *L. curtus*, *Tetrastichus julis*, *Meigenia mutabilis*, and Nematoda), and pathogenic microorganisms (*Beauveria bassiana*, *Fusarium* spp., and Microspora) that participate in the natural regulation of *L. melanopus*. The contagion by microsporidia in

case of both *Lema* species is entirely common. It was found in laboratory conditions that both *Lema* species are very sensitive to the contagion by the microsporidium *Nosema algerae* to which they react very quickly. Anderson and Paschke^[8] mention the case when ova of *L. melanopus* were parasitized by *A. flavipes*. The majority of papers mention cases where only *L. melanopus* (Linnaeus, 1785) was parasitized. Therefore our attention was focused only on *L. lichenis*. We found^[9,10] that, in our territory, the pupas (larvae) of *L. lichenis* L. are parasitized by the following species: *N. leucarthros*, *P. vibulens*, *G. instabilis*, *L. curtus*, *I. maculatus*, *Diplazon laetatorius*, *B. maculatus*, and *T. julis* (Table 1). This results from the observation of cases where *L. lichenis* L. was parasitized. Their percentage in our territory was high—56.3%; this percentage ranged generally from 34.5% (1997) to 83.6% (in the year of 2000). Individual years had great influence on it.

It was mainly *Necremnus leucarthros* (Nees, 1834) that participated in parasitizing. A similar finding was made also in the Czech Republic. From the total number of parasites parasitizing on *L. lichenis* L.,

Table 1 Spectrum of parasites on *O. lichenis* in 1995–2000

| Species | Number (in pieces) | Percentage |
|---|-----------------------|-----------------------------|
| <i>Eulophidae</i> | | |
| <i>N. leucarthros</i> (Nees, 1834) | 906 | 59.8 |
| <i>T. julis</i> (Walker, 1839) | 25 | 1.7 |
| <i>Pteromalidae</i> | | |
| <i>Pteromalus vibulens</i> (Walker, 1839) | 295 | 19.5 |
| <i>Ichneumonidae</i> | | |
| <i>B. maculatus</i> (Hellén, 1957) | 52 | 3.4 |
| <i>D. laetatorius</i> (Fabricius, 1781) | 93 | 6.1 |
| <i>G. instabilis</i> (Foerster, 1850) | 51 | 3.4 |
| <i>I. maculatus</i> (Fabricius, 1775) | 30 | 2.0 |
| <i>L. curtus</i> (Townes, 1965) | 41 | 2.7 |
| Undeterminable species | 21 | 1.4 |
| Total | 1514 | 100 |
| | | Total parasitization (%) |
| Total cocoons examined | 2691 | |
| Number of parasites hatched | 1514 | 56.3 |
| Share of parasite <i>N. leucarthros</i> (Nees, 1834), in pieces | 906 | 33.7 |
| Share of parasite <i>P. vibulens</i> (Walker, 1839), in pieces | 295 | 11.0 |

the participation of *N. leucarthros* represented 59.8% under a sexual index of 0.73. In second place was *P. vibulens* (Walker, 1839), having a participation of 19.5% at a sexual index of 0.65.

Necremnus leucarthros (Nees, 1834) (Hymenoptera, Apocrita, Chalcidoidea, Eulophidae) is a plurivoltine species that parasitizes on mature larvae or pupae of *L. lichenis* L. and on some species of the families Chrysomelidae and Curculionide. The body of the imago (Fig. 1) is dark green and lustrous; the legs and head are black. The length of the body of females is 1.9–2.1 mm; males are a little bit smaller. There are black antennas on the head having three (females) and four (males) segments with long, thin branches covered by fine hair. The forechest is very short. The webbed wings are without dark stains (Fig. 1). The postmarginal wessel of the front wing is about 1.5 times longer than a radial one. Fecundated females lay three to four eggs into the cocoon where *Oulema* is. While laying eggs, the females do not distinguish between cocoons that are parasitized or not; therefore they lay their eggs more often in the same cocoon. In our region, 8–12 eggs are hatched in one pupa. Larvae are relatively small; they are cudgel-shaped and live ectoparasitically on the body surface of the host. The larvae form pupae in the foam cocoon created by larvae of *L. lichenis* L. The larvae of *Necremnus* do not create their own cocoon. They leave the cocoon of the imago through one aperture. In our region, the parasite *N. leucarthros* participates significantly in the regulation of *L. lichenis* L. living on cereals. Such a high percentage (33.7%) was not observed in neighboring countries.

Another important parasite is *P. vibulens* (Walker, 1839) (Hymenoptera, Apocrita, Chalcidoidea, Pteromalidae). It is a plurivoltine, solitary, and probably ectophagous parasite of larvae of *O. lichenis* before pupa formation. Imago (Fig. 2) is dark green to black, and is metallically lustrous with dense net-like dotted chest. The postmarginal wessel of the front wing is

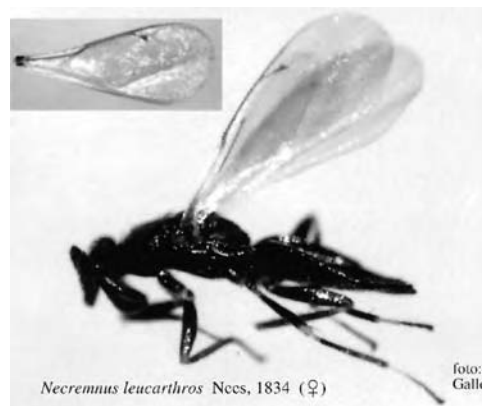


Fig. 1 Adult female (imago) of *Necremnus leucarthros* Nees, 1834, with detail of wing.

the same as the marginal one or longer. The marginal wessel of the front wing is 1.3–1.7 times longer than the radial one. The share of this parasite in parasitizing *L. lichenis* L. was between 12.5% and 15.6%. Over the period analyzed, the share of parasitizing by this species reached 11.0%.

A widespread species, *D. laetatorius* (Fabricius, 1781), had a 6.1% share of analyzed parasites. This species does not belong to the typical parasites of the *Oulema*, although its presence is observed each year. This species parasitizes mainly on species belonging to the family Syrphidae.

The parasitization of another parasite species, *L. curtus* (Townes, 1965) (Hymenoptera, Ichneumonidae), ranged between 2.0% and 6.3%. The body of the imago is dark, often nearly black. The scapus and pedicel in the bottom part of antennae are rusty red. The legs, except the pelvis (coxa), are rusty red. The sides of the middle chest are dim with fine dotting. In front of webbed wings, there is a very small triangle called areola. *Lemophagus curtus* is a solitary endogenous parasite, with mass spreading in Spain, France, Italy, and Poland, but also in other European countries. A fecundated female lays her eggs into larvae of *Lema* in such a way that she sticks her ovipositor into the body cavity. The larva, after hatching from the ovum, develops slowly and its presence has no visible influence on the health and activity of the host up to the phase where it starts to create its foam cocoon in which it should form a pupa. Then the larva of *L. curtus* leaves the body of the host and spins in its own multi-layered light yellow cocoon with a white middle belt. Here it changes, in a short time, into pupa and imago.

Over the entire period analyzed, this species had a share of 2.7% of all parasites observed.

The parasite *G. instabilis* (Foerster, 1850) (Hymenoptera, Ichneumonidae) had a share of 3.4%.

This species is polyphagous and lives hyperparasitically in ichneumons, braconids, and ground beetles; therefore pest regulation has to be evaluated negatively despite the fact that it appears in some years as a parasite of *L. lichenis* L.^[1,5] In individual years, in our



Fig. 2 Adult male (imago) of *Pteromalus vibulens* Walker, 1839, with detail of wing.

region, it participated in parasitizing on *L. lichenis* L. by 3.4–8.5%.

The remaining parasite species (*I. maculator*, *B. maculatus*, and *T. julis*) shared only weakly in the regulation of the *L. lichenis* L. population.

CONCLUSION

The most frequently occurring parasitoid of *Oulema (Lema) lichenis* (Voet, 1826) was *Necremnus leucarthros* (Nees, 1834) (Hymenoptera, Apocrita, Chalcidoidea, Eulophidae), amounting to 59.8% of total spectrum parasitoid numbers. In the future, after detailed study of biological properties and possibilities of *Necremnus leucarthros* (Nees, 1834) artificial breeding, this parasitoid could be potentially exploited in biological control against *Oulema (Lema) lichenis* (Voet, 1826). In our conditions, we have thus far recorded 8 species of parasitoids parasitizing in pupae of *Oulema (Lema) lichenis* (Voet, 1826).

REFERENCES

- Šedivý, J. Hymenopterous parasites of cereal leaf beetle. *Ochr. Rostl.* **1995**, *31* (3), 227–235.
- Balachowsky, A.S. *Entomologie appliquée a l'agriculture*; Masson et Cie Éditeurs: Sait-Germain, Paris, 1963; Tom I.
- Wetzel, Th.; Freier, B.; Heyer, W. Zur Modellierung von Befall-Schades—Relationen wichtiger Schadinsekten des Winterweizens. *Z. Angew. Entomol.* **1980**, *89*, 330–344.
- Pavlov, A. Parasites on *Oulema melanopus* L. (Col. Chrysomelidae). *Plant Sci.* **1981**, *4* (Vol. XVI), 116–123.
- Haeselbarth, E. Über einige Schlupfwespen (Hymenoptera) als Parasiten des Weizenhähnchens *Oulema lichenis* (Voet) (Coleoptera, Chrysomelidae) in Südbayern. *J. Appl. Entomol.* **1989**, *107*, 493–507.
- Bjegovič, P. Contribution to the knowledge of the Cereal Leaf Beetle (*Lema melanopa* L.) natural enemies. *Zašt. Bilja* **1971**, *114*, 173–184.
- Dysart, R.J.; Maltby, H.L.; Brunson, M.H. Larval parasites of *Oulema melanopus* in Europe and their colonization in the United States. *Entomophaga* **1973**, *18* (2), 133–167.
- Anderson, R.C.; Paschke, J.D. A biological evaluation of five European cultures of *Anaphes flavipes* (Hymenoptera, Mymaridae), an egg parasite of *Oulema melanopus* (Coleoptera, Chrysomelidae). *Entomophaga* **1970**, *15* (1), 107–120.
- Gallo, J. Occurrence of *Lema* spp. at winter wheat and parasitization of *Lema lichenis* L. Proceedings of the Slovak and Czech Plant Protection Conference, Nitra, Slovakia, Sept 3–4, 1997.
- Gallo, J. Parasites (Hymenoptera) of *Lema lichenis* on summer wheat, winter form, in Slovakia Proceedings of the Czech and Slovak Plant Protection Conference, Brno, Czech Republic, Sept 12–14, 2000.

Pathogen-Free Stock: Managing Viruses and Viroids

Rudra Singh

*Potato Research Centre, Agriculture and Agri-Food Canada,
Fredericton, New Brunswick, Canada*

INTRODUCTION

The need for virus- and viroid (small circular RNA)-free planting stock or propagating material is of paramount importance to vegetatively propagated crops, which include bulbs, corms, tubers, small fruits, tree fruits, tropical trees, woody and perennial ornamentals, and some grasses. In vegetatively propagated plants the primary source of virus and viroid infection is the infected tissue, and later infection by other viruses of such plants accumulates over generations or growing seasons. Viruses are transmitted by mechanical transfer/contact from an infected plant or transmitted from distant sources by the insects, which feed on the plants (vectors). In these plants viruses can also be spread through cultural practices (cutting, pruning, and thinning operations). The losses caused by these pathogens may not be limited to obvious symptoms of the affected plants or its harvested products, but may also extend to less visible symptoms including retardation of root development in cuttings; reduced vigor; incompatibility of stock and scions in some cultivars; reduced life span and productivity of orchards, fields, groves, and pastures. Using potato as a model for vegetatively propagated crops, attempt would be made to demonstrate the development of virus- and viroid-free stocks from infected sources to its maintenance and eventual use under commercial conditions.

POTATO CROP AS A MODEL FOR VIRUS-FREE STOCK MANAGEMENT

Potato is the fourth ranked food crop in the world today. It is a representative model for the seed management of vegetatively propagated crops, because it is susceptible to a large number of viruses and viroids. At present, 37 viruses, vectored by aphids, beetles, fungi, leafhoppers, nematodes, thrips, and whiteflies,^[1] and over 6 species of viroids^[2] are known pathogens of potato. Therefore modern management of seed potato production encompasses many facets applicable to other crops. Steps for the production of virus-free stock include 1) freeing infected propagules (tubers) from viruses; 2) rapid multiplication of the virus-free

material; 3) preventing reinfection of virus-free material under field conditions; 4) monitoring the virus levels by postharvest virus indexing; and 5) certifying the virus content of the stock at each stage of commercial potato production and export.

METHODS OF VIRUS AND VIROID ELIMINATION

When virus- and viroid-infected tubers are used for propagation, tuber-borne viruses and viroids replicate in all growing tissues (secondary infection) causing severe losses. In contrast, plants becoming infected later in the growing season (primary infection) suffer marginal losses. To minimize losses, the propagation and distribution of virus- and viroid-free stock involve the use of meristem tip culture from growing plants. In plants, apical meristems are domes of actively dividing cells, located at the apices of shoots and roots. These remain in an active state of division throughout the vegetative phase of the plant, forming new tissues and organs and have low or no virus content. However, virus content increases sharply below the apical dome. The probability of obtaining virus-free plants is inversely related to the size of the meristem used for tip culture. For potato virus X (PVX) and potato virus S (PVS) a 0.1-mm-long meristem provided less than 10% regeneration of plantlets, but of those which grew, 95% were virus-free.^[3] For potato virus Y (PVY) and potato virus A (PVA) 90% virus-free plantlets were obtained with meristem size of 0.3 mm and only 7% with meristem size of 0.8 mm.^[3]

Thermotherapy, chemotherapy, or a combination of both with in vitro propagation increases the percentage of virus-free plantlets. The percentage of PVX-free plantlets can be increased from 12.5% to 82% using 0.3-mm-long meristems and from 0% to 53% with 0.8-mm-long meristems by heating plants at 30°C from 14 to 42 days prior to meristem tip culture.^[3] Similarly, growing potato at cold temperatures (5°C) can eradicate PVA and PVY.^[3] Potato spindle tuber viroid (PSTVd)-free plantlets can also be obtained by excising meristems with 1-leaf primordium from potato plantlets, when kept at 5–6°C for 6 months or from tubers

kept at 8°C for 4 months.^[3] By growing for 14 days, in vitro nodal cuttings containing 20 mg/L of an antiviral drug, ribavirin, 18 potato cultivars and 6 *Solanum* species were freed from potato virus M (PVM), PVS, and PVX.^[3]

RAPID PROPAGATION OF VIRUS- AND VIROID-FREE MATERIAL

Virus-free plants obtained from meristem and stem-cutting cultures can easily be reinfected when grown outside. To prevent contamination, careful procedures based on the knowledge of the virus epidemiology and type of crop should be used. Virus-free nuclear stock (mother plants) should be grown in sterilized soils to avoid infection by nematodes and fungus-transmitted viruses, and in virus- and vector-free greenhouses. Material should be multiplied in insect-proof screen houses and bulk multiplication should be in isolated areas, where chances of reinfection are minimal because of the absence of virus sources and vectors.

Rapid multiplication of in vitro-derived virus and viroid-free material can be achieved by single nodal cuttings^[4] grown on MS salts medium. Alternatively, the plantlets can be grown in a greenhouse and multiplied by stem cuttings (axillary stem growth)^[5] or leaf bud cuttings (cuttings consisting of a leaf, axillary bud, and a small stem segment).^[6] Plantlets can also be transferred to the fields as transplants after hardening in the greenhouse or directly from tissue culture,^[4] or they can be used to produce minitubers (tubers produced from in vitro propagules in the greenhouse) and then planted in the field.

SEED CERTIFICATION PROGRAMS

Depending on the system, a seed certification program may regulate many aspects of virus- and viroid-free stock. It may include official recognition of cultivars, approval of the original seed source, number of generations of field growth, individual field history, isolation distance of seed fields, seed records, field inspections data, roguing of diseased or off-type plants, virus tolerances, early top-kill dates (killing the crop early to avoid virus vector peak), harvest inspection, postharvest testing, market records, certification tags, and certification program financing.^[7]

Most seed certification programs use a limited-generation production system that keeps virus incidence low by restricting continuous cultivation of a particular seed lot for a specified number of field seasons, usually 6–7 years in North America and up to 10 field seasons in Europe. Also, there are virus tolerances, e.g., a zero tolerance is for seed lots derived from

meristem tissue culture; 0 to 0.1 for the first two field generations for seed, while a much higher tolerance is accepted for commercial crops.

MANAGEMENT OF THE VIRUS-FREE CROPS IN THE FIELD

Monitoring of vector movement and dispersal in the crop is paramount in providing information on potentials for virus spread with sufficient lead time to implement management strategies to reduce tuber infection. Generally, two types of vector transmission are encountered in plants. A non-persistent vector transmission is one in which a virus could be acquired and transmitted within a few minutes, whereas a persistent vector transmission requires a few hours or days of viral incubation. The control strategies for both types of transmission are different. Insecticides can be useful for reducing in-field spread of persistent viruses especially if vectors arrive virus-free, but are not effective for non-persistent viruses. Mineral oil sprays are useful for non-persistent viruses.^[8]

In areas where soil-borne viruses are encountered, the management encompasses controlling both the virus and its soil inhabiting vectors. Potato mop top virus can be managed through exclusion and sanitation practices. Vectors can be prevented by limiting the movement from affected areas to unaffected regions through quarantine and certification of seed tubers. A promising means of reducing the incidence of tobacco rattle virus (TRV) in potato grown in affected

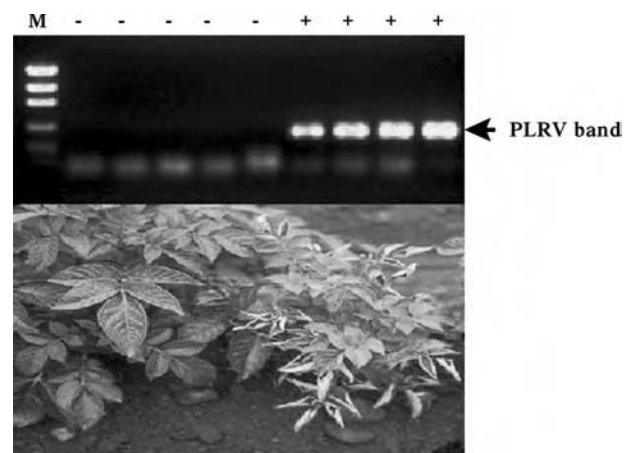


Fig. 1 Reverse-transcription polymerase chain reaction detection. Lower panel: healthy plant on the left; leafroll-infected plant on the right. Upper panel: M is size marker; – = healthy samples; + = leafroll-infected samples. Bands (arrow) are present in leafroll-infected samples but not in the healthy. (Photograph courtesy of Dr. Xianzhou Nie.)

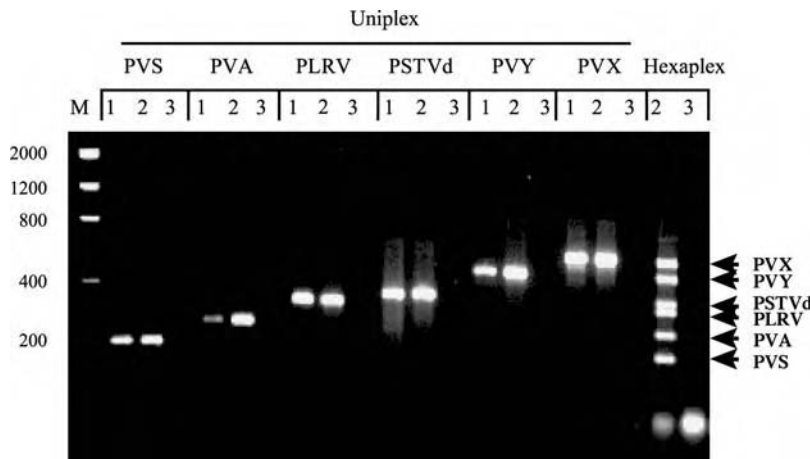


Fig. 2 Detection of single (uniplex) and multiple (hexaplex) viruses/viroids. Single detection of PVS, PVA, PLRV, PSTVd, PVY, and PVX (uniplex) and all viruses and PSTVd in one sample (hexaplex). (Reproduced with permission.)

fields is the use of rotational or cover crops, which are good hosts for the vector but poor hosts for TRV.

dures could ensure detection of trace amounts of virus and viroids, particularly essential for the first generation of meristem tip culture-derived plantlets.

NEW METHODS FOR THE SENSITIVE DETECTION OF VIRUSES AND VIROIDS

Although enzyme-linked immunosorbent assay (ELISA) has been widely used for potato virus detection, a more sensitive test such as reverse transcription-polymerase chain reaction (RT-PCR) is available for the detection of low amounts of viruses (Fig. 1). The multiplex RT-PCR, which can detect simultaneously many viruses from a single reaction sample, has advantages over ELISA and can be applied to leaf, sprouts, and dormant tubers (Fig. 2).^[9] Similarly for viroids a general method known as return-polyacrylamide gel electrophoresis (R-PAGE), applicable to many crop plants (Fig. 3), can be used.^[10] These proce-

CONCLUSION

Macroscopic plant pathogens such as bacteria and fungi are largely controlled by the use of chemical sprays. In contrast, submicroscopic viruses and viroids remain unaffected by such therapeutic treatments. Viruses and viroids are generally managed by preventative measures resulting in the reduction or elimination of infected propagules (tubers, corms, and bulbs) and virus vectors (insects transmitting viruses from plants to plants). Newer methods of virus and viroid detection, applicable to large-scale tests, can be employed for the postharvest tests or for product certification.

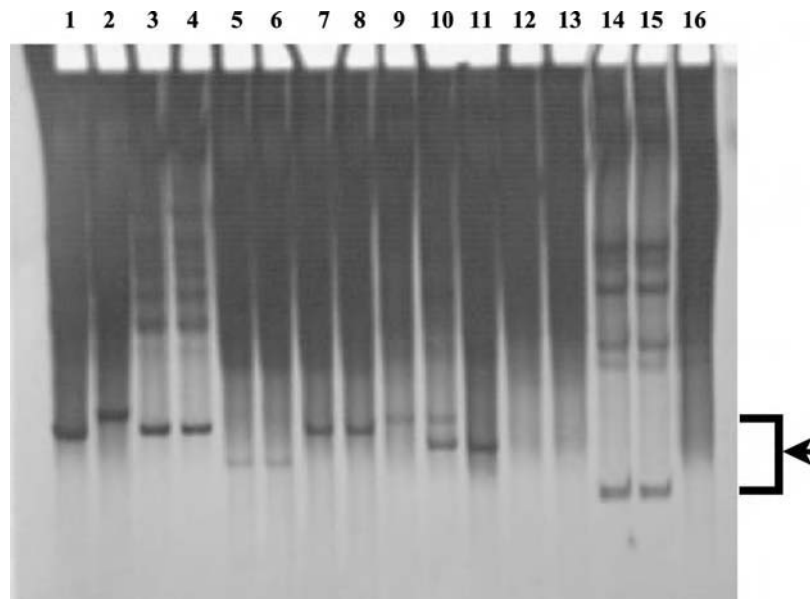


Fig. 3 Detection of viroids by R-PAGE. The lowest bands (the region of arrowhead) in each lane show the presence of specific viroids in the samples. Some have infection of two viroids.

ACKNOWLEDGMENTS

The editorial assistance of Dr. Avinash Singh is gratefully acknowledged.

REFERENCES

1. Brunt, A.A.; Loebenstein, G. The main viruses infecting potato crops. In *Virus and Viroid-Like Diseases of Potatoes and Production of Seed-Potatoes*; Loebenstein, G., Berger, P.H., Brunt, A.A., Lawson, R.H., Eds.; Kluwer Academic Publishers: Boston, 2001; 65–134.
2. Singh, R.P.; Ready, K.F.M.; Nie, X. Viroids of solanaceous species. In *Viroids*; Hadidi, A., Flores, R., Randles, J.W., Semancik, J.S., Eds.; SCIRO Publishing: Collingwood, 2003; 125–133.
3. Faccioli, G. Control of potato viruses using meristem and stem-cutting cultures, thermotherapy and chemotherapy. In *Virus and Viroid-Like Diseases of Potatoes and Production of Seed-Potatoes*; Loebenstein, G., Berger, P.H., Brunt, A.A., Lawson, R.H., Eds.; Kluwer Academic Publishers: Boston, 2001; 365–390.
4. Watad, A.A.; Sluis, C.; Nachmias, A.; Levin, R. Rapid propagation of virus-tested potatoes. In *Virus and Viroid-Like Diseases of Potatoes and Production of Seed-Potatoes*; Loebenstein, G., Berger, P.H., Brunt, A.A., Lawson, R.H., Eds.; Kluwer Academic Publishers: Boston, 2001; 391–406.
5. Cole, C.F.; Wright, N.S. Propagation of potato by stem cuttings. *Am. Potato J.* **1967**, *44* (9), 301–304.
6. Lauer, F.L. Tubers from leaf-bud cuttings: a tool for potato seed certification and breeding programs. *Am. Potato J.* **1977**, *54* (10), 457–464.
7. Gutbrod, O.A.; Mosley, A.R. Common seed potato certification schemes. In *Virus and Viroid-Like Diseases of Potatoes and Production of Seed-Potatoes*; Loebenstein, G., Berger, P.H., Brunt, A.A., Lawson, R.H., Eds.; Kluwer Academic Publishers: Boston, 2001; 421–438.
8. Ragsdale, D.W.; Radcliffe, E.B.; DiFonzo, C.D. Epidemiology and field control of PVY and PLRV. In *Virus and Viroid-Like Diseases of Potatoes and Production of Seed-Potatoes*; Loebenstein, G., Berger, P.H., Brunt, A.A., Lawson, R.H., Eds.; Kluwer Academic Publishers: Boston, 2001; 237–270.
9. Singh, R.P.; Nie, X. Multiple virus and viroid detection and strain separation via multiplex reverse transcription-polymerase chain reaction. *Can. J. Plant Pathol.* **2003**, *25* (2), 127–134.
10. Singh, R.P. Return-polyacrylamide gel electrophoresis for the detection of viroids. In *Viroids and Satellites: Molecular Parasites on the Frontier of Life*; Maramorosch, K., Ed.; CRC Press: Boca Raton, 1991; 89–107.

Pathogens of Mass Produced Natural Enemies for Biocontrol

Susan Bjornson

Department of Biology, Saint Mary's University, Halifax, Nova Scotia, Canada

INTRODUCTION

The success of an integrated pest management program is dependent on many factors, including the quality of the natural enemies that are used. In recent years, invertebrate pathogens and other microorganisms have been reported from natural enemies that are mass produced for pest control in agroecosystems or collected from the field.^[1]

Mass production systems are designed so that large numbers of natural enemies may be reared within a short period. Conditions within the rearing environment often result in overcrowding and confinement; these stresses are thought to make individuals more susceptible to disease. Furthermore, the confinement of individuals provides an opportunity for contagious pathogens to be readily transmitted from diseased to healthy individuals.^[2]

Several types of invertebrate pathogens and other microorganisms have been reported from natural enemies, including protozoa, fungi, bacteria, and viruses.^[1] The origin of many of these pathogens is unknown; therefore, the following summary describes the pathogens that may infect field-collected or mass-produced natural enemies and the effects of these pathogens on host efficacy. Although pathogens have been reported from natural enemies used for weed control, only those that are mass-produced for pest control in commercial greenhouses and on horticultural crops will be discussed.

PROTOZOAN PARASITES OF NATURAL ENEMIES

Eugregarines and microsporidia are often reported from mass-produced and field-collected natural enemies. Most eugregarine species are harmless parasites or commensals, whereas microsporidia cause chronic and debilitating disease. Following the ingestion of spores by a vulnerable host, eugregarines undergo development, partially embedding themselves into the epithelial cells of the host's intestine. Eugregarines absorb nutrients within the gut; however, the damage they cause to the intestinal epithelium is usually minimal.^[2,3] Eugregarines have been reported from several genera of coccinellids (*Adalia*, *Coccinella*, *Harmonia*, *Hippodamia*) collected from the field (cited in Ref.^[1]).

Microsporidia are parasitic, spore-forming protozoa. Each microsporidian spore contains a characteristic, tube-like polar filament (Fig. 1) and an infective sporoplasm. Although spores are commonly ingested, they may also enter the body through ovipositional wounds made by parasitoids. When spores germinate, the polar filament everts from the spore to penetrate a nearby cell and the sporoplasm is released. Microsporidia develop entirely within host cells and may be transmitted both horizontally (to healthy cohorts) and vertically (to progeny).^[3]

Steinernematid nematodes (*Steinernema carpocapsae*, *Neoplectana glaseri*) infected with microsporidia produce few infective juveniles. Partial to complete castration of both sexes may be observed and infected individuals do not live as long as healthy ones (cited in Ref.^[1]). Microsporidia reduce the fecundity and longevity of predaceous mites (*Phytoseiulus persimilis*, *Metaseiulus occidentalis*) and infected females produce fewer female progeny (cited in Refs.^[1,4]). Prey consumption of *Phytoseiulus* is significantly reduced; however, the effects of microsporidia on *Neoseiulus* have not been quantified. Microsporidia also reduce the fecundity and longevity of lacewings (*Chrysopa californica*). Although microsporidia infect predaceous coccinellids from several genera (*Adalia*, *Coccinella*, *Hippodamia*), their effects on host fitness have not been investigated (cited in Ref.^[1]).

Microsporidia are common pathogens of hymenopterous parasitoids (*Cotesia* spp., *Encarsia* nr. *pergandiella*, *Muscidifurax raptor*, *Pediobius foveolatus*, *Trichogramma* spp.). Infected parasitoids may produce fewer progeny than uninfected ones, require longer developmental times, or be unable to complete development. Parasitoids that develop within microsporidia-infected hosts may die prematurely if the host is heavily infected. Microsporidia may cause wing malformations, reduce adult emergence, or cause early mortality (cited in Ref.^[1]). Microsporidia also infect bumblebees (*Bombus occidentalis*) that are mass produced for crop pollination^[5] and have been described from an arthropod (*Tyrophagus putrescentiae*) that is used as food for mass rearing *Neoseiulus cucumeris*.^[6]

Arthropods infected with microsporidia often show no outward symptoms of disease; however, some arthropods with thin exoskeletons become milky white. This change in coloration is caused by an accumulation

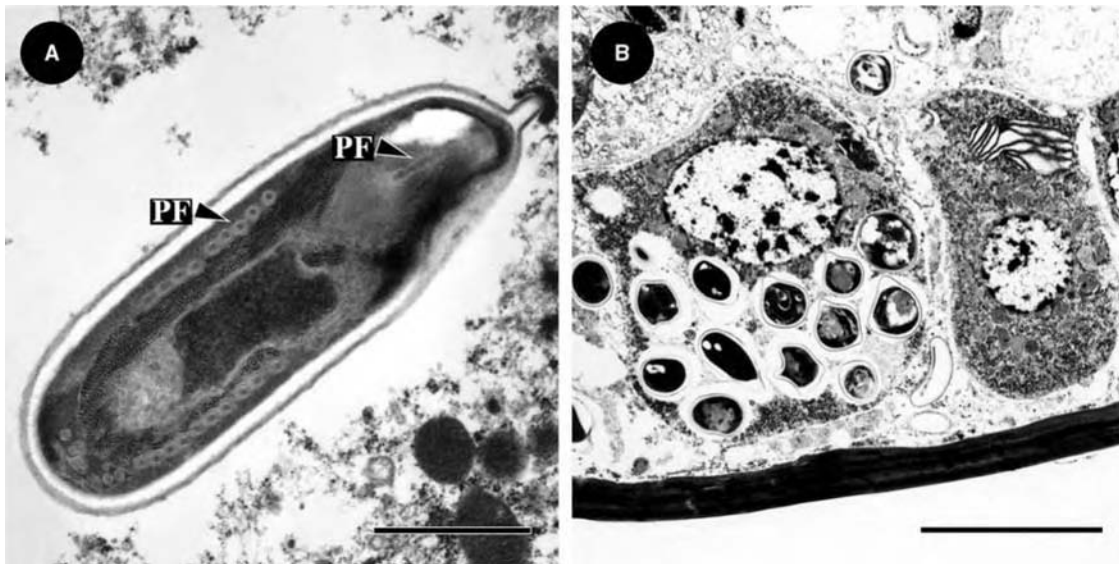


Fig. 1 Microsporidian spores from the predatory mite *P. persimilis*. (A) The characteristic tube-like polar filament (PF). (B) Mature spores inside a host cell. Scale bars, 1 and 5 μm respectively.

of spores in the body.^[2,7] Microsporidia often have subtle, but profound, effects on host fitness. They often escape notice in mass rearings; therefore, natural enemies should be examined for microsporidia on a routine basis. If detected, healthy individuals may be isolated and used to establish clean populations. Microsporidia have also been eliminated by rearing individuals at elevated temperatures or by adding antiprotozoal compounds to diets (cited in Refs.^[1,4]).

FUNGAL PATHOGENS OF NATURAL ENEMIES

Entomopathogenic fungi usually penetrate the host cuticle or enter through wounds in the integument. Fungal spores often require high temperatures and humidity to germinate. Once inside the body, the fungus competes for soluble nutrients while it invades and destroys host tissues. The presence of filamentous hyphae and fruiting structures are clear evidence of infection.^[3,7]

Entomopathogenic nematodes are prey to both predatory trapping fungi and endoparasitic fungi. The former use specialized hyphae to trap nematodes; spores of the latter adhere to the cuticle, where they germinate and penetrate the body (cited in Ref.^[1]).

The fungus *Neozygites* sp. causes mortality in the predatory mite *Neoseiulus citrifolius* but has no effect on *Neoseiulus idaeus* or *Neoseiulus limonicus* (cited in Ref.^[1]). The effects of this fungus have not been fully investigated for *Neoseiulus* species that are commonly mass-produced for pest control (for example, *N. cucumeris*, *N. californicus*, and *N. fallacis*).

Some predaceous insects are host to entomopathogenic fungi. Although the fungal pathogens *Beauveria bassiana*, *Entomophthora* sp., and *Isaria farinosa* have been isolated from adult *Aphidoletes* spp., whose predatory larvae feed on aphids, only *Beauveria* has been reported to cause adult mortality. *Beauveria* causes larval and adult mortality in green lacewings (*Chrysoperla carnea*) when they are subjected to environmental and nutritional stresses and the fungal pathogens *Verticillium lecani* and *Paecilomyces fumosoroseus* decrease fecundity, lower predatory rates, and cause mortality. *Beauveria*, a common fungal pathogen of coccinellids, affects the overwintering success of several genera (*Adalia*, *Coccinella*, *Harmonia*, *Hippodamia*). Beetles of the genus *Coleomegilla* may also be susceptible to *Beauveria*. Furthermore, the effects of other fungi (*Laboulbenia* sp., *Hesperomyces virescens*) reported from *Adalia* are not known. *Metarhizium anisopliae* and *P. fumosoroseus* cause high mortality of first-instar *Hippodamia convergens* larvae in the laboratory. *Aphidius nigripes*, an aphid parasitoid, is susceptible to *V. lecani*, a fungus used for aphid and whitefly control in commercial greenhouses. This fungus affects larval development; therefore, the timing of fungal applications is important (cited in Ref.^[1]).

Control of fungal pathogens must include altering the local temperature and humidity so as not to favor the germination of spores.^[7] The presence of nematophagous fungi in soil may reduce the survival and efficacy of entomopathogenic nematodes; therefore, more individuals may have to be released to ensure effective biological control (cited in Ref.^[1]).

BACTERIA ASSOCIATED WITH NATURAL ENEMIES

Bacterial diseases of natural enemies are rare; however, rickettsiae are intracellular symbionts that are often associated with natural enemies. Two entomopathogenic genera are recognized: *Rickettsiella* and *Wolbachia*. *Rickettsiella* are common insect pathogens that cause chronic infections in susceptible hosts. *Rickettsiella* may range in shape from small rods and cocci to large spherical forms. Crystalline bodies are often produced. Following the ingestion of rods, *Rickettsiella* infect many host tissues, causing them to lyse.^[3,7] Although *Rickettsiella phytoseiuli* is known to infect *P. persimilis*, its effect on host efficacy has not been studied (cited in Ref.^[1]).

Wolbachia are rarely pathogenic but are known to alter the reproductive biology of natural enemies by inducing parthenogenesis, feminization, male killing, or cytoplasmic incompatibilities. In the latter case, the fertilization of an uninfected female by a *Wolbachia*-infected male results in death of the developing embryo. *Wolbachia* may cause mating incompatibilities in predatory mites. Uninfected *M. occidentalis* females that mate with infected males produce few eggs and no female progeny. Although *Wolbachia* have been observed in *P. persimilis* (Fig. 2), the effects on host fitness are not known. *Wolbachia*, spiroplasmas, and other bacteria cause male killing in coccinellids of several genera (*Adalia*, *Coleomegilla*, *Harmonia*). Male embryos die early in their development, resulting in broods that are predominantly female. The causal agent responsible for male killing in *Hippodamia* has not been identified. *Wolbachia* induce parthenogenesis in several parasitoid genera (*Aphytis*, *Encarsia*, *Eretmocerus*, *Lysephlebus*, *Muscidifurax*, *Trichogramma*) but cause some parasitoids (*Nasonia* spp.) to produce only sterile male offspring or none at all (cited in Ref.^[1]).

Wolbachia may be eliminated by treating infected individuals with antibiotics (rifampicin, tetracycline) or with heat therapy (rearing at high temperatures). However, *Wolbachia* do not always have detrimental effects on host fitness and *Wolbachia*-infected parasitoids that produce high female sex ratios may be better suited for pest control (cited in Ref.^[1]).

VIRUSES AND NATURAL ENEMIES

Viruses are uncommon pathogens of natural enemies. Lysogenic phages are known to destroy bacterial symbionts that are required for normal development of steinernematid nematodes (*Photorhabdus luminescens*, *Xenorhabdus* sp.). The effects of viruses reported

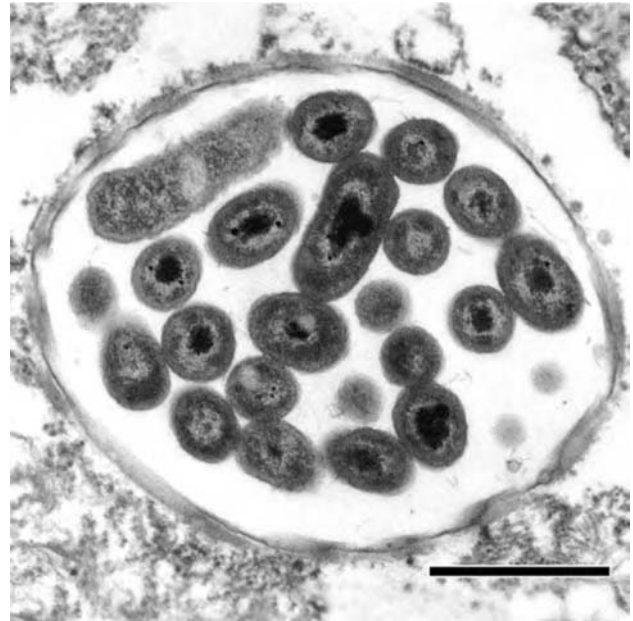


Fig. 2 *Wolbachia* in the predatory mite *P. persimilis*. Scale bar, 2 μ m.

in predatory mites (*N. cucumeris*, *P. persimilis*) and other natural enemies (*Chrysopa perla*, *Cotesia* spp.) have not been studied (cited in Ref.^[1]).

ENTOMOPATHOGENIC NEMATODES

Entomopathogenic nematodes penetrate the host cuticle or enter through natural openings, such as the spiracles.^[3] Nematodes have been reported in *Coccinella septempunctata*, whereby they invade the haemocoel, causing lower feeding and activity rates (cited in Ref.^[1]).

UNIDENTIFIED MICROBES OR DISEASE

Unidentified rickettsia-like microorganisms in *M. occidentalis* are thought to be pathogenic when mites are reared under crowded conditions in the laboratory. In some cases, rectal plugs extrude from the anus, attaching older females to their substrate. Affected individuals may show motor dysfunction, produce fewer eggs than normal, or die. Bodies of infected females become thin and transparent (cited in Ref.^[1]).

The accumulation of birefringent dumbbell-shaped crystals in *P. persimilis* causes white discoloration of the opisthosoma. Crystals are thought to be normal excretory products that are occasionally produced and excreted in excessive amounts (Fig. 3). Prominent

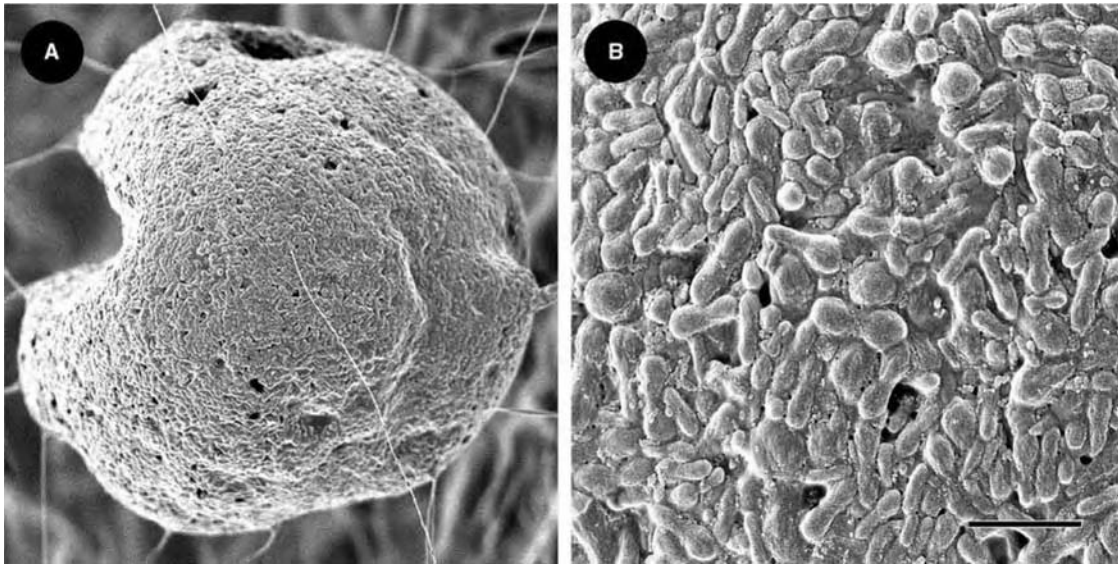


Fig. 3 (A) Fecal pellet from the predatory mite *P. persimilis* containing dumbbell-shaped crystals. (B) Higher magnification of crystals. Scale bar, 2 μ m.

symptoms have been associated with reduced fecundity and a change in foraging behavior (cited in Ref.^[1]).

An undetected pathogen is thought to cause sterility in *Eretmocerus mundus* in laboratory rearings. Sterility is associated with overcrowding of female parasitoids during oviposition. Although field-collected *Nasonia vitripennis* have been reported to produce few or no male progeny, the cause is not known (cited in Ref.^[1]).

CONCLUSIONS

Natural enemy health is paramount for the success of an integrated pest management program. The origin of many pathogens is unknown; therefore, both field-collected and mass-reared natural enemies should be examined for pathogens on a routine basis. Although some microorganisms have detrimental effects on host efficacy, others may have a more positive effect on performance. Further study is needed to fully investigate the microorganisms that occur in mass-produced natural enemies and their effects on efficacy.

REFERENCES

1. Bjørnson, S.; Schütte, C. Pathogens of mass-produced natural enemies and pollinators. In *Quality Control and Production of Biological Control Agents: Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CAB International: Wallingford, U.K., 2003; , 133–165.
2. Goodwin, R.H. Recognition and diagnosis of diseases in insectaries and the effects of disease agents on insect biology. In *Advances and Challenges in Insect Rearing*; King, E.G., Leppla, N.C., Eds.; U.S. Department of Agriculture, Agricultural Research Service, Southern Region: New Orleans, LA, 1984; 96–129.
3. Tanada, Y.; Kaya, H.K. *Insect Pathology*; Academic Press: San Diego, CA, 1993.
4. Olsen, L.E.; Hoy, M.A. Heat curing *Metaseiulus occidentalis* (Nesbitt) (Acari, Phytoseiidae) of a fitness-reducing microsporidium. *J. Invertebr. Pathol.* **2002**, *79* (3), 173–178.
5. Wittington, R.; Winston, M.L. Effects of *Nosema bombi* and its treatment fumagillin on bumble bee (*Bombus occidentalis*) colonies. *J. Invertebr. Pathol.* **2003**, *84* (1), 54–58.
6. Larsson, J.I.; Steiner, M.Y.; Bjørnson, S. *Intexta acarivora* gen. et sp. nov. (Microspora: Chytridiopsidae)—ultrastructural study and description of a new microsporidian parasite of the forage mite *Tyrophagus putrescentiae* (Acari: Acaridae). *Acta Protozool.* **1997**, *36*, 295–304.
7. Thomas, G.M. Diagnostic techniques. In *Insect Diseases*; Cantwell, G.E., Ed.; Marcel Dekker Inc.: New York, 1974; 1–48.

Pea Diseases: Ecology and Control

Bruce D. Gossen

Saskatoon Research Centre, Agriculture and Agri-Food Canada,
Saskatoon, Saskatchewan, Canada

Sheau-Fang Hwang

Alberta Research Council, Vegreville, Alberta, Canada

INTRODUCTION

Pea (*Pisum sativum* L.) is an important crop in many areas of North America. Field pea is cultivated on over a million hectares each year on the northern Great Plains, and processing and fresh-market peas are high-value, low-acreage crops in many regions. Diseases are an important constraint to production wherever the crop is grown. The objective of this article is to briefly describe the diseases of pea that are the most important constraints to production in North America. The diseases are organized alphabetically, within groups of causal agents (fungi, bacteria, viruses, and others), followed by a general section on disease management.

DOWNY MILDEW

Downy mildew (*Peronospora viciae*) develops during cool wet periods. The pathogen survives in soil, seed, and crop residue. It spreads via air-borne spores. Fluffy gray mycelia and spores develop on the undersides of leaflets, which become chlorotic on the upper surface. Systemic infection causes stunting and distortion of growth. This disease causes serious losses only in extremely wet seasons.

FUSARIUM WILT

Fusarium wilt (*Fusarium oxysporum* f. sp. *pisi*) occurs in most pea-growing regions of the world. It is spread via wind, water, contaminated seed, and farm equipment. The pathogen survives in soil by producing chlamydospores that can remain viable for many years. Repeated cropping of pea allows pathogen populations to increase, which may lead to yield losses in succeeding crops.

The pathogen at first infects roots, then progresses to stems. Symptoms include yellowing and curling of lower leaves, wilting, stunting, vascular discoloration, and plant death. Metabolites of the pathogen inhibit respiration and kill plant cells.

There are at least four economically important races of the pathogen, but further study is needed to develop

resistant cultivars. Fungicides and soil fumigants reduce disease levels, but are not cost effective.

MYCOSPHAERELLA BLIGHT (ASCOCHYTA BLIGHT COMPLEX)

Mycosphaerella blight (*Mycosphaerella pinodes*) is the dominant pathogen in the ascochyta blight disease complex, which is the most important disease of field pea on the northern Great Plains. It occurs in almost every pea field, producing dark lesions on stems, leaves, and pods, and may cause stems to collapse. Seedlings grown from contaminated seeds may not survive if lesions form at the base of the stem. Those that do survive may provide inoculum to infect the crop. Infection usually starts at lower stems and leaves. During wet periods, after the canopy closes, the disease spreads upward on foliage, stems, and pods. This results in severe loss of photosynthetic tissue, collapse of stems, and infection of seed. Lodging of weakened stems encourages further infections and hampers harvest. Seed yield can be reduced by as much as 50%, and seed infection can reduce seed quality.

Primary transmission of *M. pinodes* occurs via air-borne ascospores produced on infected crop debris (Fig. 1). Secondary transmission is from asexual spores (conidia) that are moved from plant to plant by rain splash.^[1] The other pathogens in this disease complex (*Ascochyta pisi*, *Phoma medicaginis* var. *pinodella*) do not produce ascospores.^[2] Partial resistance to mycosphaerella blight has been reported, but is not sufficient to prevent economic loss. Crop rotations of four years and incorporation of residue after harvest reduce inoculum in a field. Application of foliar fungicide is most likely to be cost effective on fresh or processing pea, because of the high return per hectare.

POWDERY MILDEW

Powdery mildew (*Erysiphe pisi*) develops only on living tissue. Epidemics usually occur late in the growing season, develop rapidly when days are warm and

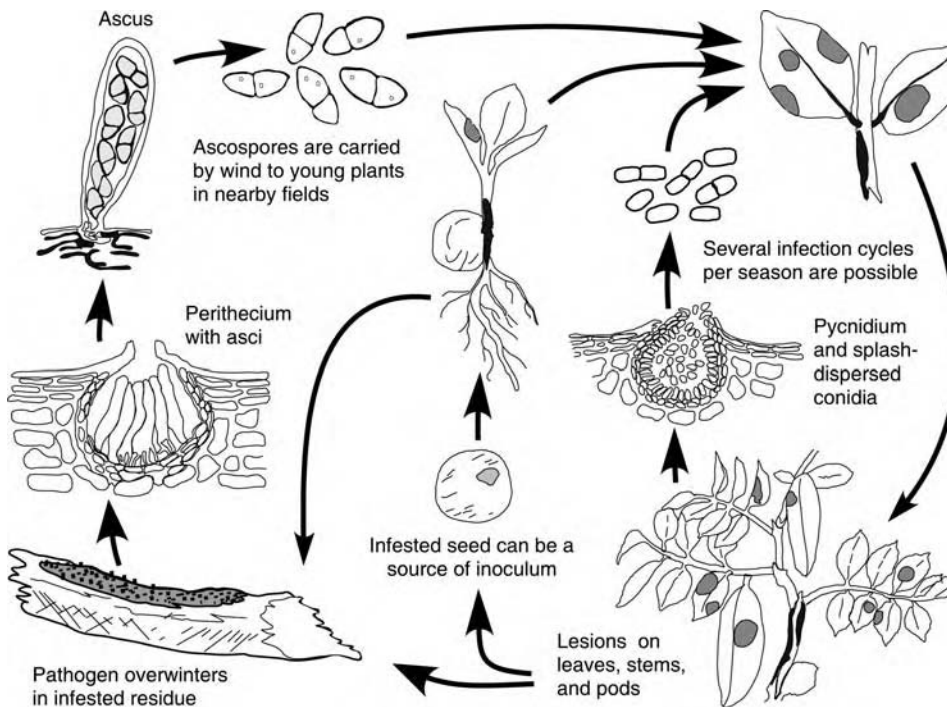


Fig. 1 Disease cycle of mycosphaerella blight caused by *M. pinodes*.

nights are cool enough to allow dew formation, and can be a serious problem in wet, low areas.

The pathogen overwinters on dead tissue. Air currents carry ascospores (primary inoculum) and conidia (secondary inoculum) to healthy plants. Leaves, stems, and pods develop fluffy white patches and eventually turn brown and die. The pathogen spreads rapidly over the entire plant. Consequently, a light infection in the lower canopy can cover the entire crop in a few days under favorable conditions. Infected plants produce fewer pods per plant and fewer seeds per pod, and yield losses of up to 50% have been reported.

Early seeding and use of early-maturing cultivars often allow the crop to escape infection. Application of foliar fungicide reduces severity, but scouting to detect initial infections is critical for successful control. Resistance is available in some commercial cultivars.

SEEDLING BLIGHT AND ROOT ROT

Although pea crops have a remarkable ability to compensate for losses in plant stand, they are generally precision-seeded because a uniform stand increases yield. Pea seed is large and has a thin seed coat, so is prone to damage during handling. Damaged seeds have a greater surface area susceptible to infection than intact seeds, and the nutrients that leak from them stimulate the growth of soil pathogens. Seedlings from damaged seed grow slowly, which increases the risk of infection. Also, the frequent occurrence of

susceptible crops in a crop rotation increases pathogen populations in the soil.

Fungicide seed treatments increase stand establishment and subsequent yield when pathogen populations are high, but should be used in conjunction with good agronomic practices, such as a three- to four-year crop rotation and selection of vigorous, disease-free seedlots.

Aphanomyces root rot (*Aphanomyces eutiches* f. sp. *pisi*) is especially destructive on wet or poorly drained soils. Soft, water-soaked lesions develop on roots and lower stems and spread rapidly. The pathogen can survive in soil for many years. Resistance or tolerance is available in some cultivars of processing pea.

Fusarium root rot (primarily *Fusarium solani* f. sp. *pisi*) occurs in most pea fields in the northern Great Plains. Reddish-brown streaks and cankers develop on roots, resulting in aboveground symptoms such as stunting, yellowing, and wilting. Seed treatment with a fungicide can increase seedling establishment and seed yield when pathogen populations are high. Resistance or tolerance is available in some cultivars of processing pea.

Pythium spp. are important pathogens of pea, particularly in poorly drained soils. *Pythium* rot reduces stand uniformity, nodulation, and nutrient utilization, resulting in decreased seed yield and quality. Seed treatment fungicides enhance seedling establishment and increase yield.^[3,4]

Rhizoctonia solani causes seedling blight and root rot in a range of crops, including pea.^[5] Affected stands

are thin and patchy. Seed treatment fungicides promote emergence and establishment.

OTHER FUNGAL DISEASES

White mold (*Sclerotinia sclerotiorum*) and gray mold (*Botrytis cinerea*) affect a range of broad-leaved crops. In pea, they are minor diseases that occasionally develop late in the season, especially in wet years under dense crop canopies.

Other fungal diseases of pea include rust (*Uromyces fabae*), Septoria leaf blotch (*Septoria pisi*), anthracnose (*Colletotrichum pisi*), Alternaria blight (*Alternaria alternata*), Cladosporium blight (*Cladosporium cladosporioides* f. sp. *pisicola*), and Thielaviopsis root rot (*Thielaviopsis basicola*). These diseases generally cause only limited damage or occur in a limited geographical region.^[6,7] The main approach for management of these diseases is crop rotation.

BACTERIAL DISEASES

Bacterial blight (*Pseudomonas syringae* pv. *pisi*) causes severe damage to pea crops around the world, especially under wet conditions. Another pathovar (pv. *syringae*) causes a similar disease called brown spot. Symptoms progress rapidly from small, water-soaked spots on the foliage to a necrotic blight that kills the plant. These pathovars overwinter mainly on seed and are readily transmitted to seedlings. It is controlled by using disease-free seeds and resistant cultivars. Pink seed (*Erwinia rhapontici*) develops under wet conditions; infected seeds are shriveled and turn intense carmine red.^[8]

VIRUSES, NEMATODES, AND OTHERS

More than 35 viruses (predominantly single-stranded RNA viruses) cause yield loss in outbreak years.^[6] A few are seed-borne, such as pea seed-borne mosaic virus, but most are transmitted by insect vectors, e.g., pea enation mosaic virus and bean leaf roll virus. Symptoms include chlorosis, mottling or other discoloration, stunting and leaf malformation, and misshapen, poorly filled pods. Those viruses that are not seed-borne generally overwinter in perennial legume crops, especially alfalfa. Disease impact can be reduced by planting healthy seed, maintaining isolation from perennial legume crops, controlling populations of vectors (cost effective only for viruses that are transmitted in a persistent manner), and use of resistant cultivars.

More than 20 genera of nematodes attack the roots of pea, but only a few are economically important.

The impact of most of these important pests is primarily because of synergistic interaction with diseases such as wilts, root rots, and viral diseases.

Nutrient deficiency or toxicity is not a frequent problem when the pea crop is grown at its preferred range of pH 6–7.^[2] Preliminary identification of nutrient problems from visual symptoms should be confirmed by soil or tissue analyses.

Hail damage is common in some regions and may result in pod splitting and reduced seed quality and yield. Immature pea plants may recover to produce good yields, but the crop is initially susceptible to infection by pathogens and saprophytes, so a fungicide application may be beneficial. Also, freezing injury can reduce seed yield and quality. In seedlings, freezing may kill the growing point, but the young plant is often able to resume growth from dormant buds. Frost injury results in fine white lesions on pods and poor quality of seed.

STRATEGIES FOR DISEASE MANAGEMENT

The economic factors that drive disease management decisions are different for dry pea and processing pea producers. For dry pea, the focus is on disease prevention, utilizing crop rotation, isolation, and vigorous disease-free seed. Availability of resistant cultivars is often limited. Seed treatments are used routinely, but in-crop disease management options may not be cost effective. Good crop rotations reduce the risk of minor diseases becoming major diseases. For processing pea, extended crop rotations may not be possible owing to the need to be near a processing plant, and cultivars are often selected to suit the processor, rather than for disease resistance. However, product quality is critical, so application of foliar fungicides may be cost effective.

CONCLUSIONS

Many pathogens attack pea, but only a few are significant constraints to production. In general, the important pathogens produce wind-borne spores that move easily between fields, or they persist in the soil and genetic resistance is not available. Disease management focuses on prevention, especially through extended crop rotation.

ARTICLES OF FURTHER INTEREST

Crop Rotation (Plant Diseases), p. 172.
Dispersal of Plant Pathogens, p. 193.
Field Crop Pest Management, p. 270.
Fungicides, p. 325.

Insect-Vectored Crop Diseases, p. 407.
Principles of Pest Management with Emphasis on Plant Pathogens, p. 666.

REFERENCES

1. Zhang, J.X.; Fernando, W.G.D.; Xue, A.G. Temporal and spatial dynamics of mycosphaerella blight (*Mycosphaerella pinodes*) in field pea. *Can. J. Plant Pathol.* **2004**, *26*, 522–532.
2. Howard, R.J.; Garland, J.A.; Seaman, W.L., Eds.; Pea. In *Diseases and pests of vegetable crops in Canada*; Can. Phytopathol. Soc. and Ent. Soc. Can.: Ottawa, ON, 1994; 202–210.
3. Hwang, S.F.; Gossen, B.D.; Chang, K.F.; Turnbull, G.D.; Howard, R.J. Effect of seed damage and metalaxyl seed treatments on pythium seedling blight and yield of field pea in Alberta. *Can. J. Plant Sci.* **2001**, *81*, 509–519.
4. Hwang, S.F.; Gossen, B.D.; Turnbull, G.D.; Chang, K.F.; Howard, R.J.; Thomas, A.G. Effects of temperature, seeding date and seed treatments on damping off and root rot of field pea caused by *Pythium* spp. *Can. J. Plant Pathol.* **2000**, *22*, 392–399.
5. Xi, K.; Stephens, J.G.H.; Hwang, S.F. Dynamics of pea seed infection by *Pythium ultimum* and *Rhizoctonia solani*: effects of inoculum density and temperature on seed rot and pre-emergence damping-off. *Can. J. Plant Pathol.* **1995**, *17*, 19–24.
6. Kraft, J.M.; Pflieger, F.L. *Compendium of Pea Diseases and Pests*, 2nd Ed.; APS Press: St. Paul, MN, 2000.
7. Kraft, J.M.; Larsen, R.C.; Inglis, D.A. Pea diseases. In *The Pathology of Food and Pasture Legumes*; Allen, D., Lenné, J.M., Eds.; CAB International: Wallingford, U.K., 1996; 325–370.
8. Xue, A.G. Diseases of pea. In *Diseases of Field Crops in Canada*, 3rd Ed.; Bailey, K.L., Gossen, B.D., Gugel, R., Morrall, R.A.A., Eds.; Canadian Phytopathological Society: Saskatoon, SK, 2003; 201–213.

Pea Insects: Ecology and Control

Juliana J. Soroka

Saskatoon Research Centre, Agriculture and Agri-Food Canada,
Saskatoon, Saskatchewan, Canada

Héctor A. Cárcamo

Lethbridge Research Centre, Agriculture and Agri-Food Canada,
Lethbridge, Alberta, Canada

INTRODUCTION

Cultivated pea, *Pisum sativum* L. ssp. *sativum*, is an annual grain legume produced for human and animal consumption and as a green manure for soil amendment. Well suited to growth in temperate areas, peas are also grown in cool areas of the subtropics and at high elevations in the tropics. Canada, France, China, and the Russian Federation produce over 60% of the 12 million tonne annual dry pea crop, while India and China produce about 58% of the nine million metric ton green pea crop.^[1]

There is little processing of the pea crop prior to utilization. Proper management of insect pests is especially critical in markets for human consumption of peas, which require a superior physical appearance of the product. The pest status of the more than 45 arthropods feeding on peas^[2,3] is determined by the feeding site on the plant and the type, magnitude, and duration of feeding of the pest. This article examines three main types of insect feeding on peas, with descriptions of major insect pests in each category.

FOLIAGE FEEDERS

Heavy defoliation by arthropods during the early seedling stage can kill pea plants and reduce stand density. At later growth stages, plants can tolerate considerable defoliation.^[4] Feeding on multiple plant parts such as foliage and root nodules, as caused by the pea leaf weevil, can decrease seed yields sharply.

Pea Leaf Weevil

Pea leaf weevil, *Sitona lineatus* L. (Coleoptera: Curculionidae) (Fig. 1), is a univoltine, common pest of pea and faba bean, clover, alfalfa, and vetch in its native Europe and the Mediterranean.^[5] It is now common in the Pacific Northwest of the U.S.A. and in Southern Alberta, Canada. In the spring, adults fly to pea fields, and females lay eggs near the base of host seedlings.

They can produce more than 3000 eggs over an extended period. Larvae feed on *Rhizobium* nodules, and undergo five instars before pupating. At maturity, larvae pupate in the soil, and the adults emerge to search for leguminaceous hosts to feed on until late in the summer. They overwinter in alfalfa or field margins.

Individual weevils consume about 6 mm² of foliage per day, leaving a characteristic scalloping on the leaf margins (Fig. 2). High infestations at the early seedling stage can severely reduce stand density. However, in most cases, plants compensate for this damage and little yield loss is experienced.^[6] Larvae, on the other hand, destroy most of the *Rhizobium* nodules (Fig. 3), which can cause significant yield loss.^[6] In the Pacific Northwest of the U.S.A., values of 0.3–1 weevil per seedling are considered sufficient to warrant the spraying of insecticides. Pesticides with a long residual action are preferred to kill weevils from more than one immigration cohort.^[7] An earlier planted trap crop, together with the use of pheromones, may concentrate weevil numbers and allow more effective insecticide control. Although the weevil is attacked by numerous natural enemies in Britain and the Mediterranean, there are no biocontrol programs in place in North America.

Other foliar-feeding pests of peas include lepidopteran larvae such as cutworms *Agrotis ipsilon* and *Xestia c-nigrum*, bud and bollworms *Helicoverpa* and *Heliothis*, armyworms *Mamestra* and *Spodoptera* spp, and beet webworm *Loxostege sticticalis*. The economic impact of these pests depends on the location, intensity, and duration of their feeding.

Leafminers

Leafminers are specialized foliage feeders that spend the larval portion of their life cycle within host leaves. Their control requires insecticides that are systemic in the plant or are absorbed through the leaf cuticle.

Pea leafminer, *Liriomyza huidobrensis* (Diptera: Agromyzidae), is an important pest of pea that has spread throughout the world within the last 25 years from its native South America. Other leafminers



Fig. 1 Adult pea leaf weevil, *S. lineatus* L. (Courtesy of H. Goulet, Agriculture and Agri-Food Canada, Ottawa, Canada.)

feeding on pea include the Old World garden pea leafminer *Chromatomyia horticola*, vegetable leafminer *Liriomyza sativae*, American serpentine leafminer *Liriomyza trifolii*, and soybean stem miner *Melanogromyza sojae*. Female leafminers puncture the leaves



Fig. 2 Crescent-shaped feeding notches on leaves of field pea caused by adult pea leaf weevil. (Courtesy of L. Dosdall, University of Alberta, Edmonton, Canada.)

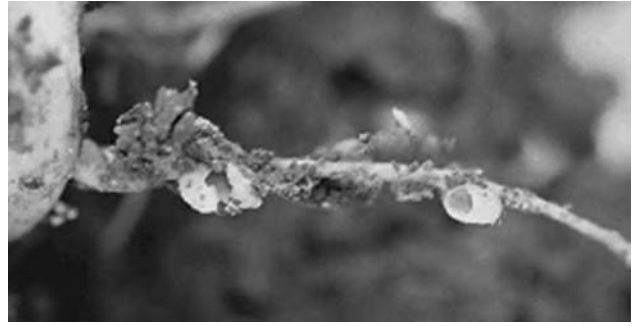


Fig. 3 *Rhizobium* sp. nodules on root of field pea hollowed out by pea leaf weevil larvae. (Courtesy of L. Dosdall, University of Alberta, Edmonton, Canada.)

of host plants, creating sites for egg-laying or feeding. Leaves that are severely mined have reduced photosynthesis, and can abscise, exposing stems, flowers, and developing fruit to heat and wind.^[2] Mining can cause delay in plant development and uneven maturity. Natural enemies of leafminers include several specific and generalist parasitoids of Agromyzid flies that usually keep pest populations under control in the field.

PHLOEM EXTRACTORS

Phloem extractors are piercing-sucking insects that feed on plant sap. The direct damaging effect of piercer-suckers such as the pea aphid *Acyrtosiphon pisum* is limited to the removal of phloem assimilates, while other insects inject a toxic saliva that can cause plant tissue necrosis and seed quality reduction.

Pea Aphid

Of Palaearctic origin, the pea aphid, *A. pisum* (Harris) (Homoptera: Aphididae), is a serious pest of pea production in North America, Europe, Asia, and Australia. *A. pisum* is oligophagous, feeding primarily on plants in the family Leguminosae. In northern latitudes, the aphid is holocyclic or sexually reproductive, overwintering as a diploid egg on perennial legumes. Winged aphids are produced in response to declining host quality or physical contact and crowding. On hosts such as pea, many generations of mainly wingless females are produced parthenogenically and viviparously (Fig. 4). At lower latitudes pea aphids may colonize suitable hosts year round and produce female offspring without mating.

Pea aphids congregate on shoot and apical meristems. Heavy aphid infestations under dry conditions cause plants to wilt and turn yellowish green, and can cause flower drop, decline in pod fill, and reduced



Fig. 4 Apterous pea aphid and nymphs. (Courtesy of M. A. H. Smith, Agriculture and Agri-Food Canada, Winnipeg, Canada.)

seed quality. Infrequently, severe stunting or plant death can occur. Pea aphids are vectors of over 30 plant pathogens, including pea leaf roll virus and verticillium wilt. Spectacular aphid population crashes can occur near the middle or end of the growing season. These crashes are caused by inclement weather such as heavy rains, declining host quality causing emigration, and mortality from a broad suite of natural enemies. Insecticide application is the principal control method, with action thresholds greater than 2–3 pea aphids per 20 cm plant tip at flowering.^[8] Plants that are infested before the flowers open often recover without loss of seed yield.

Secondary aphid pests include black bean aphid *Aphis fabae*, green peach aphid *Myzus persicae*, and cowpea aphid *Aphis craccivora*. Feeding on pea phloem through the pod wall by the polyphagous tarnished plant bug *Lygus lineolaris* (Hemiptera: Miridae) can cause “white spot,” a discoloration to the pea seed, which is especially detrimental to growers of large green peas. Feeding by the bean bug *Riptortus clavatus* (Heteroptera: Coreidae) on pea pods and seeds results in spots and discoloration, and can cause seed sterility.

SEED FEEDERS

Insects that feed on pea pods and seeds have a direct impact on seed yield and quality, and are exemplified by the pea weevil—the main pest of pea in this guild.

Pea Weevil

Primary hosts of pea weevil, *Bruchus pisorum* (L.) (Coleoptera: Bruchidae), which occurs in Europe,

Asia, North America, and Australia, are pea, bean, and maize. When temperatures reach about 20°C overwintered adults invade pea fields and feed on flower structures and pollen, a requirement for egg-laying. Females lay eggs on developing pea pods; eggs hatch, and larvae, of which there are four instars, burrow into pods and feed on developing seeds. Pupation usually occurs in the pod, with adults emerging to feed and seek overwintering sites.

Pea weevil is a major pest of legume field crops and/or legume seeds in storage.

Larval feeding on seeds reduces yield, with feeding scars and exit holes reducing pea seed quality and marketability. Seeds damaged by pea weevils can split during harvest and have lower germination. Application of contact insecticides before egg-laying is the primary control method, with economic thresholds at two or more weevils per 25 sweeps. Cultural controls include vine destruction immediately after harvest and good seed storage hygiene. Fumigation of infested seeds in storage may also be necessary. Host plant resistance holds promise, as reduced weevil viability has been found in the wild pea *Pisum fulvum*, and genetically modified pea lines that have introduced alpha amylase inhibitors provide protection against pea weevil feeding.^[9]

Other seed-feeding pests of pea include the cowpea weevil *Callosobruchus maculatae*, a secondary pest of pea seeds in the field and in storage. Pea pod borer *Etiella zinckenella* can reduce pea seed yield in India, as can the large-tailed blue butterfly *Lampides boeticus*. Stored product pests such as Mediterranean flour moth *Ephestia kuehniella* can pose a special threat to dry pea in storage.

CONCLUSIONS

A variety of insect pests attacks the world’s pea crop. Their economic impact depends on their feeding sites and habits, and on crop growth stage and health. Management of these pests necessitates knowledge of their biology and that of their natural control agents.

REFERENCES

1. FAO. FAOSTAT data 2004. Primary Crops. Agricultural production; <http://faostat.fao.org/faostat/servlet> (accessed February 2005).
2. *Crop Protection Compendium. Global Module*. CD, 2nd Ed.; CAB International, Wallingford, U.K., 2001.
3. van Emden, H.F.; Ball, S.L.; Rao, M.R. Pest, disease and weed problems in pea, lentil, faba bean and chickpea. In *World Crops: Cool Season Food Legumes*; Summerfield, R.J., Ed.; Kluwer: Dordrecht, The Netherlands, 1988; 519–534.

4. Delaney, K.J.; Macedo, T.B. The impact of herbivory on plants: yield, fitness and population dynamics. In *Biotic Stress and Yield Loss*; Peterson, R.K.D., Higley, L.G., Eds.; CRC Press: Boca Raton, Florida, 2001; 135–160.
5. Aeschlimann, J.-P. The *Sitona* (Coleoptera: Curculionidae) species occurring on *Medicago* and their natural enemies in the Mediterranean region. *Entomophaga* **1980**, *25*, 139–153.
6. Williams, L.; Schotzko, D.J.; O'Keefe, L.E. Pea leaf weevil herbivory on pea seedlings: effects on growth response and yield. *Ent. Exp. et Appl.* **1995**, *76*, 255–269.
7. Antonelli, A.; Retan, A.; O'Keefe, L.E.; Johansen, C.; *Pea Leaf Weevil: Its Biology and Control*; Extension Bulletin: Washington State University, 1985; 3 pp.
8. Maiteki, G.A.; Lamb, R.J. Spray timing and economic thresholds for the pea aphid, *Acyrtosiphon pisum* (Homoptera: Aphididae), on field peas in Manitoba. *J. Econ. Ent.* **1985**, *78*, 1442–1448.
9. Clement, S.L.; Wightman, J.A.; Hardie, D.C.; Bailey, P.; Baker, G.; McDonald, G. Opportunities for integrated management of insect pests of grain legume. In *Linking Research and Marketing Opportunities for Pulses in the 21st Century*; Knight, R., Ed.; Kluwer: Dordrecht, The Netherlands, 2000; 467–480.

Peach Diseases: Ecology and Control

Alan R. Biggs

*Kearneysville Tree Fruit Research and Education Center, West Virginia University,
Kearneysville, West Virginia, U.S.A.*

INTRODUCTION

Peaches and nectarines (*Prunus persica* L. Batsch.) are grown in the temperate zones of the northern and southern hemispheres of earth. The total peach production in the world is about 10 million tons, second of the temperate tree fruits (apple is the first). The highest number of peach orchards is around the Mediterranean Sea, with Italy accounting for about 19% of world production. While peach production is decreasing in the United States and is stable in the European Union, it is increasing in China and in South America, particularly in Chile. In most countries, the main problems in the peach industry include low quality of the fruits, high production costs, international competition, and overproduction.

There are a number of diseases that commonly occur annually, in both commercial and backyard plantings of peaches and nectarines. These diseases do not infect at the same time but appear in a fairly regular sequence, depending on the weather and the development or phenology of the host, beginning at dormancy and continuing until the fruits are harvested. Consequently, a season-long program for disease management is often necessary to harvest a high percentage of useable fruits. The diseases that are common on peach and nectarine include peach leaf curl, brown rot, perennial canker, and peach scab. Other locally important diseases include bacterial spot, shot hole, powdery mildew, and root infections that are caused by an assortment of fungi and nematodes (Table 1). Peaches and nectarines serve as hosts also to viruses and phytoplasmas, some of which are capable of inducing significant disease if not properly managed. In general, weather conditions greatly influence both the occurrence and severity of plant diseases. As a result, diseases are generally most difficult to control in the years that have high temperature, high humidity, and abundant rainfall and cloud cover.

Stone fruit diseases can be effectively managed through the combination of the cultural control practices, sanitation, resistance of plants, and fungicide sprays. This integrated approach to disease control minimizes the reliance upon one type of control over the others and usually produces fruits of high quality.

Cultural methods include maintaining tree vigor by proper planting, fertilizing, and pruning and by following general practices that help to minimize tree stress. Sanitation involves pruning and removing affected or dead portions of the tree and removing diseased foliage or fruit, which are often important sources of inoculum for the next season. Resistance involves selection and planting of varieties that have genetic resistance to specific diseases. This effectively reduces or eliminates occurrence of the disease in question. Proper selection, timing, and application of fungicide sprays are important. Thorough coverage of all parts of the tree is necessary. Information about the plant hosts and diseases, dosage rates, days-to-harvest intervals, and safety precautions can be found on the fungicide label.

PEACH LEAF CURL

Peach leaf curl (Fig. 1) is caused by the fungus *Taphrina deformans*. Symptoms occur in the spring season, and infection of emerging leaves is favored by cool, wet spring weather. The fungus causes a thickened, reddish-purple discoloration of developing leaves. These leaves become puckered, primarily along the midvein, and appear distorted and stunted. As these leaves with apparent symptoms age, they become yellow to brown and drop from the tree. A second crop of leaves is subsequently produced, which is not affected by the disease. The fungus overwinters in the buds and infects newly developing leaves as the buds begin to swell in the spring. Infection occurs only during a relatively short time period as fungal spores are washed onto developing leaves by rain. Although this fungus rarely infects or causes symptoms on fruit, several years of uncontrolled heavy leaf infection can weaken the tree and effectively reduce its life span.

Leaf curl is effectively controlled by properly selected and timed fungicide sprays. In fact, a single application can provide nearly total control of the infection. To be effective, the fungicide application must be made before the buds begin to swell—this can be done in late fall, after the leaves have fallen, or in early spring before the buds swell.

Table 1 Common diseases and their causal organisms found on peaches and nectarines

| | |
|--------------------------------|---|
| Brown rot | <i>Monilinia</i> spp. |
| Leaf curl | <i>T. deformans</i> |
| Scab | <i>C. carpophilum</i> |
| Peach canker | <i>Leucostoma</i> spp. |
| Shot hole | <i>Wilsonomyces carpophilus</i> |
| Powdery mildew | <i>Podosphaera leucotricha</i> <i>Sphaerotheca pannosa</i> |
| Rust | <i>Tranzschelia discolor</i> |
| Bacterial canker | <i>Pseudomonas syringae</i> |
| Bacterial spot | <i>Xanthomonas arboricola</i> pv. |
| Crown gall | <i>Agrobacterium</i> sp. |
| Root rot | <i>Armillaria</i> spp. <i>Phytophthora</i> spp. |
| Root-knot nematodes | <i>Meloidoyne</i> spp. |
| Ring nematode | <i>Criconemella</i> sp. |
| Lesion nematode | <i>Pratylenchus</i> sp. |
| Dagger nematode | <i>Xiphinema</i> sp. |
| Virus and phytoplasma diseases | |

BROWN ROT

Brown rot (Fig. 2) is caused by the fungi *Monilinia fructicola* and *M. laxa*. It is the most common and destructive disease of peaches and nectarines worldwide. The disease is especially severe in warm, wet, and/or humid weather. The brown rot fungus can cause blossom blight, twig blight, twig canker, and fruit rot. Infected blossoms wilt, shrivel, and die, after

becoming covered by molds that are tan to grayish in color. Infection can spread to the twig and form a brownish, oval canker. These cankers can expand and eventually girdle the twig, causing the terminal growth to wither and die. The disease first appears on the fruit, when they begin to mature and ripen, as a small, circular, brown spot that increases rapidly in size and eventually results in the rot of the entire fruit. Under humid conditions, tan to gray, powdery tufts of fungus appear on the surface of the fruit, a characteristic diagnostic symptom of this disease. Fruit decay is often not apparent on green fruit. Fruits that are wounded (by insects, mechanical injury, bird pecks, etc.) are more readily infected than nonwounded fruit. Rotted fruit may fall or persist on the tree where they harden. Hardened, infected fruits are termed “mummies.” The fungus overwinters in fruit mummies on the tree or ground and in twig cankers. In spring, the fungus produces two types of spores; one type (conidia, from asexual reproduction) is produced on the surface of cankers and mummified fruits on the tree, and the other type (ascospores, from sexual reproduction) is produced in mummified fruits on the ground. Both spore types can cause infection under warm, moist conditions.

Sanitation is essential to control brown rot. Mummified fruit that remain on the tree should be removed and destroyed and all dead and/or cankered twigs should be pruned and removed from the vicinity of the tree or planting. In addition, mummified fruit on the ground should be raked and removed and/or cultivate the land under the tree, to prevent the formation of ascospores and conidia on the mummies in spring. During harvest, care should be taken to avoid bruises, punctures, or tears in the skin of mature fruit to



Fig. 1 Swollen and discolored leaf tissue that is characteristic of peach leaf curl caused by *T. deformans*.



Fig. 2 Tan-colored spore production on peach fruit that is infected by the brown rot fungus *M. fructicola*.

prevent sites for potential infection. Along with the sanitation program, a season-long spray program is usually necessary for an effective control of brown rot.

PERENNIAL CANCKER

Perennial canker (Fig. 3) is caused by two related fungi *Leucostoma cinctum* and *L. personii*. It is known also as Valsa canker, Cytospora canker, and peach canker. This disease is a particular problem in the northern areas of the temperate region because it is more serious on trees that have been weakened or damaged by cold

temperatures. These fungi cause cankers that first appear as sunken areas in the wood, which exude considerable gum. Cankers are generally oval in shape and are often, but not always, surrounded by a roll of callus at the canker margins. Cankers increase in size annually, and if no breakage occurs in the initial stages, they often completely girdle a limb or an entire trunk and cause the death of that part of the plant. The fungi overwinter in cankers or on dead wood. Conidia and ascospores are produced and carried by splashing and wind-driven rain and/or wind. Infection requires moisture and takes place through a variety of different avenues, including winter injuries, pruning cuts



Fig. 3 Cankered branch tissues characteristic of peach canker disease caused by *L. personii*.



Fig. 4 Heavily infected peach fruit showing separate and coalesced greenish lesions caused by the scab fungus *C. carpophilum*.

(especially stubs from improper pruning), mechanical damage, insect punctures, and leaf scars.

An integrated approach is necessary to manage this disease. Pruning of trees should be delayed until March or April or later, if possible, to promote early healing of wounds. Any cankers or dead wood should be pruned and either destroyed or removed from the vicinity of the planting. All efforts to avoid mechanical damage, to maintain the vigor of the tree by proper application and timing of fertilization, and to protect the tree from insect and other injuries should be made. Protective fungicide sprays have not been found to be of value.

PEACH SCAB

Peach scab (Fig. 4), or ink spot, is caused by the fungus *Cladosporium carpophilum*. It is most often a problem in warm, wet weather following the shuck-split stage of development. Although the fungus infects leaves and twigs, disease symptoms are most often observed on the fruit. Early infection appears on the fruit as small, circular, green spots that are concentrated at the stem end. The infections develop slowly; therefore the spots usually do not appear until the fruits are half-grown. Older lesions appear as dark-green to black, velvety blotches that, as they coalesce, often cause cracks in the skin and flesh. Infections on twigs and leaves are inconspicuous and appear as green to reddish-brown superficial lesions. The fungus overwinters in lesions on twigs, and in spring it produces spores called conidia. The conidia are washed and splashed by rain to twigs, fruits, and leaves where they cause new infections. There is a gap of 40–70 days from the time the

conidia land on the fruit to the time of appearance of symptoms of the disease. So, the disease can go unnoticed until the fruits are well grown.

Sanitary practices such as pruning and removing twigs that show the symptoms of the disease help to reduce the overwintering inoculum. However, scab is primarily controlled with fungicide sprays.

CONCLUSIONS

The ecology and the management of diseases that affect peach and nectarine plants have been briefly presented. Four diseases have been discussed. However, there is a great diversity in the disease-causing microorganisms that are potentially present in any particular situation. The incidence and severity of diseases can vary widely, depending on the local conditions. Similarly, the occurrence of the diseases discussed, as well as the additional ones listed in Table 1, can vary by the location. For example, peach canker is less common in areas that experience infrequent winter injury. Also, the powdery mildews are more common in drier locations, and scab is more important in warm, humid production regions.

BIBLIOGRAPHY

- Fideghelli, C.; Della Strada, G.; Grassi, F.; Morico, G. The peach industry in the world: present situation and trend. *Acta Hort.* **1998**, *465*, 29–40.
- Ogawa, J.M., Zehr, E.I., Bird, G.W., Ritchie, D.F., Uriu, K., Uyemoto, J.K., Eds.; *Compendium of Stone Fruit Diseases*; APS Press: St. Paul, 1995; 98 pp.

Peanut Diseases: Ecology and Control

Barbara B. Shew

Department of Plant Pathology, North Carolina State University,
Raleigh, North Carolina, U.S.A.

Farid Waliyar

ICRISAT, Patancheru, Hyderabad, Andhra Pradesh, India

INTRODUCTION

Peanut or groundnut, *Arachis hypogaea* L., is a legume grown in countries having warm climates (Table 1). Peanut is grown for local consumption in much of Africa and Asia. Intensive management is a major requirement in areas where peanut is grown as a cash crop. Light, well-drained, slightly acid soils are ideal for peanut production. Peanut fruits (pods) are produced underground on specialized structures called pegs. Short season (duration) type peanut fruits can mature in 90 days, whereas long duration types may require 150 days or more for maturity. During this period, foliage, roots, stems, pods, and seeds are attacked by an array of pathogens (Table 2). Available host resistance generally is moderate at best; thus, disease management depends on integration of resistance with cultural and chemical methods of control (Table 2).

LEAF SPOTS

Early leaf spot caused by *Cercospora arachidicola* and late leaf spot caused by *Cercosporidium personatum* (syns. *Phaeoisariopsis personata*, *Passalora personata*) are the major foliar diseases that affect peanuts worldwide. The teleomorphs, *Mycosphaerella arachidis* and *M. berkeleyi* have scarcely been observed.^[1,2]

Conidia are disseminated by wind, rain, and insects. Epidemics develop when prolonged periods (>8 hr/day) of leaf wetness and temperatures of 18–28°C occur over several days. Latent periods range from 10 to 21 days, and several disease cycles are possible in a typical growing season. Heavy leaf spot infections lead to severe defoliation and pod shedding. Yield losses of 50% or more are common wherever disease-control measures are not taken.

Conidia produced on crop debris or volunteer plants are the primary inoculum. The fungi lack specialized survival structures, have little saprophytic ability, and attack only peanuts. Rotation therefore delays onset of leaf spot epidemics, but alone is not sufficient to prevent losses.

Foliar fungicides typically are applied every 14 days, starting from 30–45 days after planting, for up to eight sprays per season. Chlorothalonil, pyraclostrobin, trifloxystrobin, azoxystrobin, tebuconazole, and propiconazole provide excellent control. Copper- and sulfur-based fungicides are inexpensive but less effective. Fungicides may be mixed or alternated to increase efficacy and prevent fungicide resistance.

The number of sprays can be reduced by using weather-based advisories. Advisories use temperature and moisture (humidity, irrigation, rainfall) to identify periods when sprays are needed.^[1] The number of sprays also can be reduced by timing sprays to coincide with key developmental stages.^[3]

Most cultivars have low resistance to leaf spots. Progress in breeding for resistance has been slowed by association of resistance with low yield, small pod size, and late maturity, and by barriers to introgression of resistance from wild *Arachis* species. However, some moderately resistant cultivars have been developed and further progress is expected.^[4,5]

RUST

Peanut rust, caused by *Puccinia arachidis*, occurs worldwide. It is economically important in Africa, Asia, and Central and South America. Sporadic outbreaks occur in the southernmost United States. The uredinial stage is predominant and teliospores are observed infrequently. Pycnidia and aecia have not been described, nor have alternate hosts.^[1,2]

Windblown urediniospores can be dispersed over long distances to initiate epidemics. Prolonged leaf wetness and temperatures of 20–30°C favor rust infections. With latent periods of 12–21 days and copious spore production, rust can spread very rapidly, leading to yield losses of more than 50% on unprotected plants.

Urediniospores are short lived, especially in cold or dry conditions. Thus, short fallow periods, rotation, and destruction of volunteers can prevent or delay onset of epidemics. Most fungicides that control leaf spots are also effective against rust. More frequent

Table 1 Total peanut production, planted area, and yield in leading peanut producing countries (2001/2002)

| Country | Production (1000 metric tons) | Area (1000 ha) | Yield (kg/ha) |
|-----------|----------------------------------|-------------------|------------------|
| China | 14,415 | 4990 | 2890 |
| India | 7600 | 8200 | 930 |
| U.S.A. | 1940 | 571 | 3400 |
| Nigeria | 1490 | 1220 | 1220 |
| Indonesia | 990 | 635 | 1560 |
| Senegal | 903 | 920 | 980 |
| World | 33,109 | 22,760 | 1450 |

Statistics of oilseeds, fats, and oils. USDA-NASS Agricultural Statistics 2003. http://usda.gov/nass/pubs/agr03/03_ch3.pdf (accessed November 2004).

applications may be needed to control the more destructive rust epidemics.

Moderate to high partial resistance to rust has been identified in peanut germplasm. Resistance to rust, early leaf spot, and late leaf spot is inherited independently, and all may be needed for successful foliar disease management. Several sources of partial resistance to rust and late leaf spot have been identified for use in areas where both are endemic.^[6]

STEM ROT

Sclerotium rolfsii, anamorph of the basidiomycete *Athelia rolfsii*, causes a disease called stem rot, which is also known as southern stem rot, Sclerotium wilt, or white mold. Sclerotia in soil germinate in response to moisture and volatile stimulants to infect stems, crowns, pegs, and pods. Signs and symptoms include white mycelium, tan to brown sclerotia, shredded brown lesions, wilting, and death. Rank vegetative growth in warm, wet weather encourages plant-to-plant spread. In dry seasons, the fungus is most active underground and damage may not be apparent until digging.^[1,2]

Although *S. rolfsii* has a very broad host range, it does not attack grains and many other grasses. Rotations with vegetables should be avoided. Burying infested residue by deep plowing traditionally has been recommended for stem rot control. However, conservation tillage does not appear to increase disease severity.

Some triazoles and strobilurins used to manage leaf spot also control stem rot. These fungicides are applied during the period critical for stem rot development, about 45–90 days after planting. Flutolanil also is highly active against stem rot.

CYLINDROCLADIUM BLACK ROT

Cylindrocladium parasiticum, anamorph of the ascomycete *Calonectria ilicicola*, causes a severe root, peg,

and pod rot known as *Cylindrocladium* black rot (CBR). It affects the economy of peanut cultivation in the U.S.A. and Australia, and is also found in Japan, India, and Brazil.

Because the fungus is active in cool soils, most infections occur early in the growing season. Above-ground yellowing and wilting usually do not become obvious until plants near maturity; brick-red perithecia also may be present. Microsclerotia are released into soil as the crop residues degrade and serve as inoculum in subsequent crops. Very low rates of seed transmission can occur.

The partial CBR resistance available in several cultivars is most effective when combined with other management practices. Cotton and grains are excellent rotation crops. Soybean is a host of *C. parasiticum*, where it causes red stem disease, and must be avoided. Bedding the soil before planting maximizes warming and reduces infections. Delayed planting likewise is encouraged.^[1,5]

In severely infested fields, growers may use the fumigant metam sodium before planting. It is applied by chisel directly under the row at rates (48 kg/ha) that reduce, but do not eradicate, *Cylindrocladium* and nematode (Table 2) populations.

GROUNDNUT ROSETTE

Groundnut rosette disease was first reported in 1907 from Tanganyika (Tanzania), and has since been reported throughout sub-Saharan Africa. In 1975, an epidemic in northern Nigeria caused an estimated loss of \$250 million. Similarly, an epidemic in eastern Zambia in 1995 caused losses of \$4.89 million.

Groundnut rosette disease has a complex etiology involving aphid vectors (primarily *A. craccivora*) and three agents: 1) *Groundnut rosette assistor virus* (GRAV), a luteovirus that causes symptomless infection and is transmitted by aphids; 2) *Groundnut rosette virus*

Table 2 Summary of important peanut diseases and their control^a

| Disease | Pathogen(s) | Areas of economic importance | Epidemiology | Principal control measures |
|---|---|------------------------------|--|--|
| Early leaf spot | <i>Cercospora arachidicola</i> | Worldwide | Polycyclic; airborne | Fungicides; rotation; resistance (low to moderate) |
| Late leaf spot | <i>Cercosporidium personatum</i> (syn. <i>Phaeoisariopsis personata</i> ; <i>Passalora personata</i>) | Worldwide | Polycyclic; airborne | Fungicides; rotation; resistance (low to moderate) |
| Rust | <i>Puccinia arachidis</i> | Asia, Africa, South America | Polycyclic; airborne | Fungicides; resistance (low to moderate); rotation; fallow |
| Stem rot | <i>Sclerotium rolfsii</i> | Worldwide | Primarily monocyclic; soilborne | Rotation; fungicides; cultural |
| Seedling diseases ^a | <i>Aspergillus niger</i> ; <i>Rhizoctonia</i> spp.; <i>Pythium</i> spp.; <i>Fusarium</i> spp. | Worldwide | Monocyclic; seed- and soilborne | Fungicides; rotation; cultural |
| Limb and pod rot ^a | <i>Rhizoctonia</i> spp. | U.S.A. | Primarily monocyclic; soilborne | Rotation; fungicides; cultural |
| Sclerotinia blight ^a | <i>Sclerotinia minor</i> | U.S.A., Australia | Primarily monocyclic; soilborne | Resistance (moderate); fungicides; cultural |
| Nematodes ^a | <i>Meloidogyne arenaria</i> ; <i>M. hapla</i> ; <i>M. javanica</i> ; <i>Pratylenchus brachyurus</i> ; <i>Belonolaimus lognicaudatus</i> | Worldwide | Polycyclic; soil borne | Rotation; granular insecticide/nematicides; resistance (<i>M. arenaria</i> , low to high) |
| Cylindrocladium black rot | <i>Cylindrocladium parasiticum</i> | U.S.A., Australia | Monocyclic; soilborne; seedborne | Resistance (moderate); rotation; fumigation; cultural |
| Aspergillus and aflatoxin | <i>Aspergillus flavus</i> ; <i>A. parasiticus</i> | Worldwide | Pre- and postharvest; air- and soilborne | Cultural; resistance; biocontrol |
| Tomato spotted wilt/peanut bud necrosis | <i>Tomato spotted wilt virus</i> ; <i>peanut bud necrosis virus</i> (Tospovirus) | U.S.A., Asia, Africa | Primarily monocyclic; thrips vector | Resistance (moderate); cultural |
| Groundnut rosette | <i>Groundnut rosette virus</i> + <i>groundnut rosette assistor virus</i> + sat RNA | Africa, Asia | Aphid vector | Resistance (moderate to high); cultural |

^aFor information on diseases not discussed in this article, readers are referred to Ref.^[1].

(GRV), an umbravirus that causes symptomless infection and is aphid transmitted only in the presence of GRAV and sat RNA; and 3) sat RNA, which is aphid transmitted in the presence of GRAV and GRV and requires GRAV and GRV to cause symptoms. *Groundnut rosette virus* is mechanically sap transmissible, as is sat RNA in the presence of GRV. *Groundnut rosette assistor virus* is not mechanically transmissible.^[7]

The predominant symptom types of groundnut rosette disease are “chlorotic” and “green” rosette.

Disease can be diagnosed in the field based on symptoms or the agents can be detected by serological and/or molecular methods. Early sowing and maintaining uniformly dense stands greatly reduce disease incidence.

All available rosette resistant lines of cultivated peanut are susceptible to GRAV. Resistance to GRV was reported in 130 long duration lines and 20 short duration lines.^[8] Some wild *Arachis* accessions are reported to be resistant to all three components of

groundnut rosette disease.^[7] Putative transgenic plants carrying coat protein and replicase genes have been produced at the ICRISAT center in India and will be tested soon.

TOMATO SPOTTED WILT AND PEANUT BUD NECROSIS

Tomato spotted wilt virus (TSWV) or the closely related tospovirus *Peanut bud necrosis virus* (PBNV) are found worldwide. These viruses cause devastating diseases of peanut. The symptoms of TSW include stunting, bud necrosis, ring patterns, necrotic blotches, and wilting. The first symptoms of PBN usually appear on newly formed leaves as chlorotic spots that may develop into chlorotic and necrotic rings. These leaflets become flaccid and droop before dying, leading to typical necrosis of the terminal buds. Stunting and proliferation of axillary shoots are common symptoms of PBN after systemic spread.^[1,9]

Tomato spotted wilt virus is vectored by thrips (e.g., *Frankliniella fusca*, *F. occidentalis*), which transmit the virus in a persistent manner. *Thrips palmi* is the main vector of PBNV in India. Many plant species are hosts of the viruses and their thrips vectors. Thrips carrying the virus move from crop or weed hosts to emerging peanut seedlings. Severe infections can kill young plants, but apparent increases in disease incidence are observed throughout the growing season. This reflects symptom development from early infections; secondary infection is limited. Seed transmission does not occur.

Because of the wide host range of the viruses and vectors, rotation and weed control are not effective for disease management. Close seed spacing within and between rows reduces the damage caused by the disease, as does the use of in-furrow insecticides. Early and very late planting should be avoided in temperate areas. In semi-tropical areas, sowing in mid-June with the onset of rains limits PBN. Similarly, in the post-rainy season, PBN incidence is lower in November-sown crops than in December-sown crops.

Mechanisms of host resistance are not well understood, but several field resistant cultivars have been developed. Resistance must be combined with other management approaches to minimize losses. Published TSW risk indices help growers to choose successful combinations of management approaches.^[9]

ASPERGILLUS AND AFLATOXIN

Aflatoxins are toxic, carcinogenic, teratogenic, and immuno-suppressive substances produced when toxic strains of the fungi *Aspergillus flavus* and

A. parasiticus grow on peanut, maize, cotton, and many other agricultural commodities. Aflatoxins are highly toxic to livestock and are well recognized as a cause of liver cancer. About 4.5 billion persons living in developing countries are chronically exposed to aflatoxins.

Many countries have placed limits on the levels of aflatoxins permissible in imported peanuts and peanut products. The European Union has a limit of 4 µg/kg total aflatoxins. Many other countries have limits ranging from 10 to 30 µg/kg.

Infection of peanut by *Aspergillus* occurs under both preharvest and postharvest conditions. Preharvest infection, and consequent aflatoxin contamination, is most important in the semi-arid tropics, especially when the end-of-season drought occurs.^[10,11] Damage by soil insects also can lead to aflatoxin contamination. Postharvest contamination occurs when insect damage or excess moisture during storage leads to rapid fungal growth and production of aflatoxins.^[1] Studies in Africa have shown that storing peanuts in their pods causes greater insect damage and subsequent fungal colonization.

There is a good correlation between drought at the end of the season and aflatoxin contamination.^[10] Any crop management practice that can improve water retention at the end of the season or avoid drought is likely to reduce aflatoxin contamination.

Several genotypes have been identified as resistant or tolerant to *A. flavus* infection and aflatoxin production.^[11] Some accessions also have shown resistance to seed infection. Some of these lines were used extensively in breeding programs, and several drought tolerant lines with high yield potential and high resistance/tolerance to *A. flavus* infection and aflatoxin contamination have been produced.

Several biocontrol agents have been reported to control aflatoxins in peanut. A commercial formulation of nontoxicogenic *Aspergillus* strains has been developed in the United States.^[12] In vitro studies at ICRISAT showed *Trichoderma viride* and *T. harzianum* as potential biocontrol agents. The greatest reduction in aflatoxin contamination (>97%) was observed with a biocontrol agent (*Trichoderma viride*), gypsum + biocontrol agent (*T. viride*) + compost treatment application.

CONCLUSIONS

Diseases are major constraints to peanut production worldwide. Managing these diseases requires integration of host resistance, cultural methods, and chemical inputs. Significant progress has been made in the development of high quality, well-adapted cultivars with resistance to multiple diseases. Widespread

availability of such cultivars will maximize yields while minimizing inputs in all production areas.

REFERENCES

1. Kokalis-Burelle, N.; Porter, D.M.; Rodriguez-Kabana, R.; Smith, D.H.; Subrahmanyam, P. *Compendium of Peanut Diseases*, 2nd Ed.; APS Press: St. Paul, 1997.
2. Porter, D.M.; Smith, D.H.; Rodriguez-Kabana, R. Peanut plant diseases. In *Peanut Science and Technology*; Pattee, H.E., Young, C.T., Eds.; Amer. Peanut Res. Educ. Soc.: Yoakum, TX, 1982; 326–410.
3. Waliyar, F.; Adamou, M.; Traore, A. Rational use of fungicide applications to maximize peanut yield under foliar disease pressure in West Africa. *Plant Dis.* **2000**, *84* (11), 1203–1211.
4. Wynne, J.C.; Beute, M.K.; Nigam, S.N. Breeding for disease resistance in peanut (*Arachis hypogaea* L.). *Ann. Rev. Phytopathol.* **1991**, *29*, 279–303.
5. Shew, B.B.; Beute, M.K.; Stalker, H.T. Toward sustainable peanut production: progress in breeding for resistance to foliar and soilborne pathogens of peanut. *Plant Dis.* **1995**, *79* (12), 1259–1261.
6. Mehan, V.K.; Reddy, P.M.; Subrahmanyam, P.; McDonald, D.; Singh, A.K. Identification of new sources of resistance to rust and late leaf spot in peanut. *Int. J. Pest Manage.* **1996**, *42* (4), 267–271.
7. Naidu, R.; Kimmins, F.M.; Deom, C.M.; Subrahmanyam, P.; Chiyembekeza, A.J.; vander Merwe, P.J.A. Groundnut rosette: a virus affecting ground production in sub-Saharan Africa. *Plant Dis.* **1999**, *83* (8), 700–709.
8. Subrahmanyam, P.; Hildebrand, G.L.; Naidu, R.A.; Reddy, L.J.; Singh, A.K. Sources of resistance to groundnut rosette disease in global groundnut germplasm. *Ann. Appl. Biol.* **1998**, *132* (3), 473–485.
9. Culbreath, A.K.; Todd, J.W.; Brown, S.L. Epidemiology and management of tomato spotted wilt in peanut. *Ann. Rev. Phytopath.* **2003**, *41*, 53–75.
10. Hill, R.A.; Blankenship, P.D.; Cole, R.J.; Sanders, T.H. Effects of soil-moisture and temperature on preharvest invasion of peanuts by the *Aspergillus-flavus* group and subsequent aflatoxin development. *Appl. Environ. Microbiol.* **1983**, *45* (2), 628–633.
11. Waliyar, F.; Ba, A.; Hamma, H.; Bonkougou, S.; Bosc, J.P. Sources of resistance to *Aspergillus flavus* and Aflatoxin contamination in groundnut genotypes in West Africa. *Plant Dis.* **1994**, *78* (7), 704–708.
12. Dorner, J.W.; Cole, R.J.; Blankenship, P.D. Effect of inoculum rate of biological agents on pre-harvest aflatoxin contamination of peanuts. *Biol. Control.* **1998**, *12* (3), 171–176.

Pear Insects: Ecology and Control

David R. Horton
Thomas R. Unruh

USDA-ARS, Wapato, Washington, U.S.A.

INTRODUCTION

Over 15 million metric tons of pears were produced worldwide in 2004, of which China, the United States, and Italy produced about 60%, 6%, and 5%, respectively, of the total.^[1] In the United States, pears are grown commercially in nine states, with three western states (Washington, Oregon, and California) accounting for more than 90% of the pear acreage.^[2]

Pears are attacked by a number of arthropod pests that reduce grower return either by directly damaging the fruit or by reducing yield. This suite of pests includes two primary taxa, codling moth (*Cydia pomonella*) and the pear psyllids (*Cacopsylla* spp.), and other secondary pests distributed among several orders (see Table 1). This entry will briefly summarize the biology of the primary arthropods attacking pears, with emphasis on the North American and western European faunas. The discussion of biology will be followed by an overview of control approaches, with sections on insecticide programs, mating disruption, and biological control.

PEST ARTHROPODS

Codling moth (*Cydia pomonella*) is the most serious pest of pome fruits in the temperate fruit growing regions of the world except in Japan, Eastern China, and Western Australia.^[3] The insect has one to five generations per year, depending on climate. It overwinters as a mature larva in its cocoon spun in a crevice on or near the tree. In spring, moths fly a few weeks after pears bloom, mate and lay eggs singly on or near the fruit. The first instar larvae move to fruit and burrow into its pulp to feed. This pest originated in the Palearctic and has been in the United States for more than 200 years. Other important internal pear-fruit feeders include the pear codling moth (*Cydia pyrivora*), the Manchurian codling moth (*Cydia inopinata*), the peach fruit moth (*Carposina sasakii*), and the Oriental fruit moth (*Grapholita molesta*). The Asian species, *C. inopinata* and *C. sasakii*, are significant quarantine concerns for Europe, the Americas, and New Zealand.

Several species of Psyllidae (*Homoptera*) in the genus *Cacopsylla* are important pests of pears (see

Table 1). Taxonomic status of the west Palearctic species of pear-feeding psyllids was clarified by Burckhardt and Hodkinson.^[4] The North American representative, *C. pyricola*, is a European species introduced into the eastern United States in the early 1800s;^[5] it now occurs in all pear growing regions of North America. Horton^[5] summarized the biology of pear psylla in North America. The pest has two to five generations per year, depending upon latitude. Egg-laying by the overwintered generation begins in late winter, well before foliage eggs appear. The eggs are deposited directly into the wood. Eggs of the summer generation are deposited into flower or leaf tissues. There are five nymphal instars. Beating trays are used to monitor adult psylla, while eggs and nymphs are monitored by taking spur, shoot, and leaf samples. Damage is caused by the feeding activities of nymphs. The nymphs produce large amounts of honeydew, and contact of this exudate with the fruit surface produces dark blotches or streaks, resulting in downgrading of the fruit. The damage is worsened by a sooty mold fungus that colonizes the honeydew and also marks the fruit. High densities of nymphs may additionally cause reductions in yield and fruit size.

Secondary pests include representatives from several arthropod groups, including leafrolling lepidoptera, mealybugs, aphids, a leaf curling midge, scale insects, and mites (see Table 1). Taxonomic composition of the leafroller complex varies regionally in North America, but the complex will commonly include the obliquebanded leafroller and an *Archips* species. The spider and rust mites are important secondary pests, and the rust mite especially can cause significant fruit damage and downgrading by its direct feeding on the fruit.

PEST CONTROL

Insecticidal Control

Insecticidal control of pests remains the standard for pear production in Europe and North America. The earliest sprays occur in late winter while the tree is still dormant, and consist of mineral oil often in combination with an insecticide. These sprays are directed

Table 1 Major pest groups attacking pear

| Type of damage | Pests |
|---|---|
| Internal fruit feeders | Lepidoptera: Tortricidae <i>Cydia pomonella</i> (codling moth), <i>Cydia pyrivora</i> * (pear codling moth), <i>Cydia inopinata</i> * (Manchurian codling moth), <i>Grapholita molesta</i> (Oriental fruit moth), <i>Carposina sasakii</i> * (peach fruit moth) |
| External fruit, leaf feeding | Lepidoptera: Tortricidae <i>Archips rosanus</i> (European leaf roller), <i>Archips argyrospila</i> (fruittree leafroller), <i>Archips podana</i> (large fruit-tree tortrix), <i>Pandemis pyrusana</i> (pandemis leafroller), <i>Pandemis heparana</i> (dark fruit-tree tortrix), <i>Adoxophyes orana</i> * (summerfruit tortrix), <i>Choristoneura rosaceana</i> (obliquebanded leaf roller), <i>Argyrotaenia citrana</i> (orange tortrix), <i>Argyrotaenia velutinana</i> (redbanded leafroller), <i>Platynota stultana</i> (omnivorous leafroller), <i>Cacoecimorpha pronubana</i> (carnation tortrix), <i>Syndemis musculana</i> (autumn leafroller) Acari: Eriophyidae <i>Epirimerus pyri</i> (pear rust mite) Acari: Tetranychidae <i>Tetranychus urticae</i> (two-spotted spider mite), <i>Panonychus ulmi</i> (European red mite) |
| Leaf feeders which damage fruit with their exudates | Homoptera: Psyllidae <i>Cacopsylla pyricola</i> , <i>Cacopsylla pyri</i> *, <i>Cacopsylla pyrisuga</i> *, <i>Cacopsylla bidens</i> Homoptera: Pseudococcidae <i>Pseudococcus maritimus</i> (grape mealybug) Homoptera: Aphididae <i>Dysaphis plantaginea</i> (rosy apple aphid), <i>Dysaphis piri</i> * (pear bedstraw aphid) |
| Leaf damage; rarely fruit or flowers | Diptera: Cecidomyiidae <i>Dasineura pyri</i> (pear leaf curling midge) Acari: Eriophyidae <i>Phytoptus pyri</i> (pear leaf blister mite) |
| Damage stems and occasionally fruit | Homoptera: Diaspididae <i>Diaspidiotus perniciosus</i> (San Jose scale), <i>Diaspidiotus piri</i> * (pear scale) Homoptera: Aphididae <i>Eriosoma lanigerum</i> (woolly apple aphid) |

Many of these species also attack other pome, stone and small fruits. This list is far from exhaustive, especially for the leaf feeding pests. (*not found in the Americas.)

at pear psylla, scale, and eggs of the European red mite. Subsequent pre-bloom sprays include oil supplemented with an insecticide directed at pear psylla. Early season control is critical for psylla management, as the pest shows explosive egg-laying potential during this period, and the natural enemies that attack psylla are generally not abundant enough at this time of year to control the pest.

Biorational alternatives for early season control of psylla include mineral oil sprays to interfere with oviposition, insect growth regulators to prevent egg hatch and for disrupting the molting process, and Surround (kaolin clay) to repel the adult psylla and interfere with oviposition. Kaolin may be applied several more times during the season, but is typically used one to three

times up to bloom. At bloom, a second application of an insecticide or growth regulator may be used for psylla. Applications of abamectin or neonicotynl insecticides directed at pear psylla may be made following petal fall.

Insecticide sprays for codling moth are implemented following petal fall, consisting generally of azinphos-methyl, phosmet, or a similar broad-spectrum insecticide. These products severely disrupt natural enemy populations. One or two additional sprays of broad spectrum insecticides may be used against the codling moth in its first flight, through June, and two or three additional sprays may be directed against its second and subsequent generations. Less disruptive chemicals, including neonicotinyls, growth regulators, and spinosad,

Table 2 Pear pests thought to be at least partially regulated by parasitoids, and genera of parasitoids responsible

| Pest group | Parasitoid family and genera |
|--------------------|--|
| Pear psyllids | Encyrtidae: <i>Trechmites</i> , <i>Prionomitus</i> |
| Leafroller complex | Braconidae: <i>Apanteles</i> , <i>Oncophanes</i> , <i>Orgilus</i> , <i>Macrocentrus</i> , <i>Meteorus</i> , <i>Microgaster</i> Ichneumonidae: <i>Apophua</i> , <i>Diadegma</i> , <i>Glypta</i> , <i>Itopectis</i> , <i>Triclistus</i> Eulophidae: <i>Colpoclypeus</i> , <i>Sympiesis</i> Tachinidae: <i>Actia</i> , <i>Nilea</i> , <i>Nemorilla</i> , <i>Pseudoperichaeta</i> |
| Scale insects | Aphelinidae: <i>Encarsia</i> , <i>Aphytis</i> |
| Mealybugs | Encyrtidae: <i>Acerophagus</i> , <i>Anagyrus</i> , <i>Leptomastix</i> |
| Aphids | Aphelinidae: <i>Aphelinus</i> Aphidiidae: <i>Aphidius</i> , <i>Ephedrus</i> , <i>Lysiphlebus</i> |

may be substituted for the broad-spectrum products. A biopesticide, granulosis virus, is also used against the codling moth by some fruit growers, and may be an important component of the pest control program in organic orchards. Summer insecticide sprays for secondary pests such as leafrollers, mites, and mealybug are necessary in many orchards. These sprays may disrupt biological control.

Mating Disruption Control

Pheromone-based mating disruption of the codling moth represents an effective alternative or supplement

to conventional insecticides for its control.^[6] This approach consists of dispensing synthetically produced sex pheromone at such a high rate or at enough point sources that the pheromone interferes with the males' ability to locate females. A formulation consisting of the main pheromone component, codlemone, together with some minor components, has been used successfully worldwide. Adequate control of the codling moth may require both mating disruption and insecticides, especially in settings where two or more generations of the moth occur. The use of mating disruption, in combination with selective insecticides, may allow substantial reduction of pesticide use.^[7]

Table 3 Common predatory arthropods in North American and European pear orchards

| Taxon | Family: Important genera | Presumed prey |
|--------------|--|---|
| Acari | Phytoseiidae: <i>Amblyseius</i> , <i>Neoseiulus</i> , <i>Typhlodromus</i> Stigmaeidae: <i>Zetzellia</i> Anystidae: <i>Anystis</i> | Mites |
| Dermaptera | Forficulidae: <i>Forficula</i> | Generalists |
| Heteroptera | Anthocoridae: <i>Anthocoris</i> , <i>Orius</i> Miridae: <i>Campylomma</i> , <i>Campyloneura</i> , <i>Deraeocoris</i> , <i>Heterotoma</i> , <i>Orthotylus</i> , <i>Pilophorus</i> , <i>Phytocoris</i> Nabidae: <i>Nabis</i> | Mites, aphids, psyllids, mealybugs, eggs of Lepidoptera |
| Thysanoptera | Aelothripidae: <i>Aelothrips</i> Thripidae: <i>Scolothrips</i> | Mites, thrips |
| Neuroptera | Chrysopidae: <i>Chrysopa</i> , <i>Chrysoperla</i> Hemerobiidae: <i>Hemerobius</i> , <i>Micromus</i> , <i>Sympherobius</i> | Aphids, psyllids, mealybugs |
| Diptera | Syrphidae Cecidomyiidae: <i>Aphidoletes</i> | Aphids |
| Coleoptera | Coccinellidae: <i>Adalia</i> , <i>Chilocorus</i> , <i>Coccinella</i> , <i>Cryptolaemus</i> , <i>Harmonia</i> , <i>Hippodamia</i> , <i>Scymnus</i> , <i>Stethorus</i> Carabidae | Aphids, psyllids, mites, mealybugs, scale insects codling moth |
| Araneae | | Generalists |

(From Refs.^[9,10] unpublished sampling studies.)

BIOLOGICAL CONTROL

Parasitoids

Many parasitoids attack pear pests and in some cases their activities help maintain low pest densities. The pest species that appear to be most affected by the activities of parasitoids are listed in Table 2, along with the parasitoids attacking these pests. Notable among these pests is the San Jose scale which is often heavily parasitized by the wasps *Encarsia perniciosi* and *Aphytis* spp. Parasitism coupled with dormant oil sprays often provides complete scale control in pears. Similarly, in the absence of disruptive insecticide sprays, the following pests may be heavily attacked by their associated parasitoids: pear psylla, especially by *Trechnites* spp.,^[8] grape mealybug, especially by *Acerophagus*, *Anagyrus*; and *Leptomastix*. Leafrollers may also suffer significant parasitism from a large complex of parasitoids (see Table 2).

Predators

A taxonomic variety of mostly generalist predatory arthropods occurs in pear orchards (see Table 3). Important predators of pear psylla include especially several genera of true bugs, but also Coccinellidae, Chrysopidae, and Forficulidae. The codling moth is relatively safe from predators due to its feeding habits within the fruit, but adults may be susceptible to spiders, and large larvae in search of pupation sites and in their cocoons are vulnerable to ground beetles, spiders, and birds. The best summary and taxonomic list of predators in pear orchards is that by Solomon et al.^[9] for European orchards. A similar review for North American orchards is long overdue. Quantitative estimates of impact by natural enemies attacking pests in pear orchards are mostly lacking. Much of the evidence available suggesting that predators contribute to biological control of pests in pear orchards is correlative, consisting primarily of observations that pest numbers decline in orchards having high populations of predators. There is considerable room for quantitative research on biological control in pear orchards.

CONCLUSIONS

Pear orchards support diverse communities of pest and beneficial arthropods. Pest control focuses especially upon managing an internal fruit feeder, the codling

moth, often with use of broad-spectrum insecticides. These non-selective products are highly disruptive to natural enemies, leading often to problems caused by secondary pests, including pear psylla, mites, leafrollers, and mealybugs. The secondary pests, in turn, may require control using chemical sprays. Biorational approaches consisting of mating disruption for the codling moth coupled with narrow spectrum insecticides and biological control for secondary pests are being used. These selective approaches may be highly effective, but can be challenging to implement because of the need for intensive monitoring of the arthropod community, and the need for use of correctly timed insecticide applications.

REFERENCES

1. Anonymous. *FAS Quarterly Reference Guide to World Horticultural Trade: World Production, Supply, and Distribution of Key Products*; Circular Series FHORT 1-04; U.S. Department of Agriculture, Foreign Agricultural Service: Washington, DC, 2004; 28–30.
2. Anonymous. *Noncitrus Fruits and Nuts: 2003 Summary*; U.S. Department of Agriculture, National Agricultural Statistics Service: Washington, DC, 2004; 60–63.
3. Barnes, M.M. Tortricids in pome and stone fruits: codling moth occurrence, host race formation, and damage. In *Tortricid Pests, Their Biology, Natural Enemies and Control*; van der Geest, L.P.S., Evenhuis, H.H., Eds.; Elsevier: Amsterdam, 1992; 313–327.
4. Burckhardt, D.; Hodkinson, I.D. A revision of the west Palearctic pear psyllids (Hemiptera: Psyllidae). *Bull. Entomol. Res.* **1986**, *76*, 119–132.
5. Horton, D.R. Monitoring of pear psylla for pest management decisions and research. *Integ. Pest Mgmt. Rev.* **1999**, *4*, 1–20.
6. Howell, J.F.; Knight, A.L.; Unruh, T.R.; et al. Control of codling moth, *Cydia pomonella* (L.) in apple and pear with sex pheromone-mediated mating disruption. *J. Econ. Entomol.* **1992**, *85*, 918–925.
7. Hilton, R.J.; Westigard, P.H. Pear pest management using codling moth mating disruption and horticultural spray oil. *Acta Hort* **1994**, *367*, 410–417.
8. Unruh, T.R.; Westigard, P.H.; Hagen, K.S. Pear psylla. In *Biological Control in the Western United States*; Nechols, J.R., Andres, L., Beardsley, J.W., Goeden, R.D., Jackson, G., Eds.; DANR Publications, University of California: Davis, 1995; 95–100.
9. Solomon, M.G.; Cross, J.V.; Fitzgerald, J.D.; et al. Biocontrol of pests of apples and pears in northern and central Europe—3. Predators. *Biocont. Sci. Tech.* **2000**, *10*, 91–128.
10. Beers, E.H.; Brunner, F.J.; Willett, J.M.; Warner, G.M. *Orchard Pest Management: a Resource Book for the Pacific Northwest*; Good Fruit Grower: Yakima, 1993.

Pecan Insects: Ecology and Control

Marvin K. Harris

Department of Entomology, Texas A&M University, College Station, Texas, U.S.A.

INTRODUCTION

The pecan, *Carya illinoensis*, is a hickory native to North America whose ancestry dates back some 60 million years before present.^[1] Pecan is the most important uncultivated horticultural plant in North America,^[2] with about 50% of current agricultural production in the native range coming from autochthonous trees. Mature pecan is a large (30 m+ height), long-lived (200 yr+) tree that has a 200 plus days growing season. Pecan domestication to improve nut production began about 150 years ago with the advent of grafting this species. Domestication accentuated interest in pecan insects and their control as more human resources were invested in this gift of nature. Virtually all pecan insects of today share this origin, and investigating their ecology and control also provides insights into their phylogenetic origins.^[3] Thus, much of what is known of pecan and pecan insects stems from a curiosity driven by the agribusiness embedded in a natural ecosystem. This is contrary to most agricultural plants, whose production centers are far removed from their aboriginal homes, and whose arthropod complexes are an assemblage of coevolved and recently adapted species.^[4] Pecan is unique. Domestication is occurring with a largely coevolved arthropod complex, for which the pecan has already evolved defenses for survival. Arthropod control for nut production can be integrated with these natural defenses to replace deficiencies resulting from differences between agronomic requirements and biological success. Modern pecan insect control programs were developed by investigating pecan-insect interactions to determine which posed a threat to production, followed by studies on how best to mitigate that threat, consistent with all other production needs.^[5]

PECAN INSECT ECOLOGY

The pecan insect complex consists of hundreds of biologically fascinating species. This treatment will focus on a few species of agricultural importance determined by: 1) leaf and nut life table studies,^[6-8] 2) an epidemic history,^[9] and 3) being routinely targeted for pesticide sprays by producers.^[10] Pecan leaf feeders include defoliators, leafminers, shoot feeders, and phloem

feeders. Leaf life table studies show that <10% of the foliage is lost in an average season to these herbivores.^[8] Epidemic history shows that phloem feeders experience modest outbreaks annually and some other species do defoliate large areas every few decades. Some producers annually target the black-margined pecan aphid, *Monellia caryella*, based on sighting honeydew, and the black aphid, *Tinocallis caryae-foliae*, based on leaf damage. The leaf life table studies and extensive investigations of phloem feeders indicate that pecan leaf feeders seldom pose a threat to production and should only be targeted if economic thresholds are exceeded.^[11] Overreliance on insecticides to eliminate evidence of foliar insects, typically results in resistance, resurgence, and pollution.^[9,10] Additionally, *M. caryella* outbreaks are typically curtailed by an intrinsic foliar defense bolstered by natural enemies that remain, if undisturbed, to prevent other more insidious pests from causing more serious damage.^[12] The role of this aphid appears to be to reestablish waning densities of natural enemies in the natural system. The black aphid typically infests foliage in the shaded canopy in late summer and affected leaves discolor and dehisce. Black aphid outbreaks can spread from this relatively unproductive area of the canopy to the more productive sun leaves on the exterior, unless controlled by natural enemies. The black aphid typically affects foliage from limbs that will be naturally shed in a few years time as the tree continually grows to produce a higher canopy. This places the black aphid in the role of an impatient detritivore with little effect on pecan fitness in the natural system. Pecan nut feeders include the monophagous pecan nut casebearer, *Acrobasis nuxvorella*, and pecan weevil, *Curculio caryae*. The nut life table studies show both species can seriously reduce nuts surviving to dehiscence (harvest). Epidemic history and pesticide targeting confirm their pest status.^[13] Nut mortality from the casebearer, a facultative nut feeder, is inversely related to crop load.^[14] Casebearer poses no threat to a bumper crop, but will devastate a low yet harvestable nut set. Apparently, casebearer capacity for epidemic on a bumper crop is limited by a combination of natural enemies, environmental factors, and seasonal host susceptibility. The role of casebearer in the natural system appears to be to remove all nuts in years of low production, while being innocuous in bumper

crop years. The bi-triennial pecan weevil obligatorily attacks mature nuts of *Carya* and, given unlimited food, densities will rise (rate of increase of about fivefold/generation) to the carrying capacity of the environment killing all nuts.^[15] Each adult pecan weevil destroys about 15 nuts. The role of the pecan weevil in the natural system appears to be that of a formidable predator capable of severely reducing plant fitness. The lack of an annual life cycle indicates that the pecan weevil is adapted to a host that fluctuates in nut production through time and that bears few if any nuts following a bumper crop year. Indeed, pecans in the natural system synchronously produce nuts every two to seven years, averaging large crops every three to four years.^[16] This usually allows the wild pecan to escape the pecan weevil in time. This potential for escape is enhanced by the pecan nut casebearer accentuating barrenness by also removing immature nuts in years of low production, which limits nut availability for all later season nut feeders.

In summary, the ecology of pecan insects appears strongly influenced by their pecan host. Foliage feeders are limited by intrinsic confrontational pecan defenses like tannins in leaves and bioassociations as illustrated above with *M. caryella*, in addition to density-dependent interactions with natural enemies. The rich assemblage of pecan foliage feeders appears to coexist with pecan in niches that minimally impact pecan fitness. Pecan insect nut feeders appear limited by a combination of food availability and natural enemies. The pecan strategy of nut defense using escape in time is not very compatible with needs of production agriculture.

PECAN INSECT CONTROL

Management strategies in pecan to aid agricultural production must consider which insects to target and how to manage them in a 200 plus days growing season without creating even worse collateral effects.^[17] The fundamental principle underlying management is to only introduce overt management when a damaging pest is about to occur in damaging numbers. Regular surveillance of foliage allows foliage feeders to be detected and their densities monitored before economically important numbers occur. Threshold densities that threaten production have been developed and are used as a trigger to implement needed control measures, typically a pesticide spray.^[18] Selective materials, when available, are recommended to conserve natural enemies. Recognition of the cost/benefits of *M. caryella* has engendered an increased grower tolerance for some honeydew accumulation and the limited loss in production, if any, that this may entail. However, pecan varieties differ in susceptibility to this aphid,^[19] and producers of “Cheyenne” typically

experience higher aphid densities for longer periods and may thus be compelled to control them. Growers of autochthonous pecans and less susceptible varieties like “Pawnee” would rarely benefit from treatment of this aphid. Growers of improved pecans often begin to encounter black aphid as expanding tree canopies densely shade lower limbs (two decades or so after the orchard is established). The problem is exacerbated when needed treatments earlier in the season reduce natural enemies. Pruning shaded limbs and thinning trees usually removes this unproductive niche and minimizes the black aphid problem. Most growers need not treat for foliage feeders in most years. Regular surveillance of developing nuts and insect nut feeders allows incipient damaging infestations to be detected and controlled. Casebearer management is aided by a prediction model, a pheromone monitoring protocol, and a sequential sampling plan, and provides control with a single well-timed treatment when needed.^[10] Similarly, pecan weevil management is aided by monitoring adult emergence from the soil and nut susceptibility to infestation (post-gel stage) to decide if and when control is needed. The annual production expected in managed pecan systems typically requires two to four pesticide treatments annually to prevent economically important nut loss primarily from insect nut feeders.^[13] This results in pesticide intervention providing about three weeks of residual protection in the growing season. The remaining 90% of the time, natural forces are relied upon to defend the crop.

CONCLUSIONS

The natural defenses of pecan against insects are escape in space and time, confrontation, accommodation, and bioassociations.^[4] The great diversity of insect species associated with pecan contains few that routinely threaten plant fitness or agricultural production. Natural selection has apparently provided conditions where most species exist by parsimonious use of yield-sensitive tissues or by utilizing alternative resources. Recognition of these natural defenses allows the producer to only bolster those needed for commercial production, while minimally perturbing the natural system. Insistence upon insect-free appearance of pecan canopies results in insecticide resistance, resurgence, and unnecessary pollution.^[17] The escape in time and bioassociation with casebearer that the pecan uses for defense against nut feeders in the natural system is incompatible with the needs of production agriculture. The advent of selective pesticides against lepidopterans (*Bacillus thuringiensis* endotoxins, tebufenozide, spinosad, etc.) allows conservation of many natural enemies compared to the organophosphate, carbamate, and pyrethroid alternatives.^[17] Casebearer control, when

needed in early season, is minimally disruptive when these selective materials are used. The material of choice for pecan weevil control is carbaryl or, less desirably, an organophosphate or a pyrethroid. Carbaryl requires fewer treatments, appears to provide better control, and is thought less disruptive of natural enemies than the alternatives. Barring the emergence of a new pest species, or regulatory loss of carbaryl, pecan insect control appears sustainable through the remainder of this decade.

REFERENCES

1. Thompson, T.E.; Grauke, L.J. Pecans and hickories (*Carya*). In *Genetic Resources of Temperate Fruit and Nut Crops*; Acta Horticulturae, Number 290-XVIII; Int. Soc. Hort. Sci.: Wageningen, 1991; 839–904.
2. Brison, F.R. *Pecan Culture*; Capitol Press: Austin, Texas, 1974.
3. Harris, M.K. Pecan domestication and pecan arthropods. In *The Entomology of Indigenous and Naturalized Systems in Agriculture*; Harris, M.K., Rogers, C.E., Eds.; Westview Press: Boulder, Colorado, 1988; 207–225.
4. Harris, M.K. Arthropod-plant interactions in agriculture, emphasizing host plant resistance. In *Biology and Breeding for Host Plant Resistance to Arthropods and Pathogens in Agricultural Plants*; Harris, M.K., Ed.; Agric. Comm. Texas A&M University: College Station, Texas, 1980; Misc. Publ., 1451, 23–51.
5. Harris, M.K. Integrated pest management of pecan. In *Handbook of Pest Management in Agriculture*, 2nd Ed.; Pimentel, D., Ed.; CRC Press Inc.: Boca Raton, Florida, 1991a; Vol. III, 691–700.
6. Harcourt, D.G. Crop life tables as a pest management tool. *Can. Entomol.* **1970**, *102* (8), 950–955.
7. Harris, M.K.; Cutler, B.L.; Ring, D.R. Pecan nut loss from pollination to harvest. *J. Econ. Entomol.* **1986**, *79* (6), 1653–1657.
8. Ring, D.R.; Harris, M.K.; Olzak, R. Life tables for pecan leaves in Texas. *J. Econ. Entomol.* **1985**, *78* (4), 888–894.
9. Harris, M.K. Pecan arthropod management. In *Pecan Husbandry: Challenges and Opportunities*; Wood, B.W., Payne, J.A., Eds.; First National Pecan Workshop, Proc. U. S. Dept. Agric., Agric. Res. Serv., ARS-96, 1991b; 6–15.
10. Harris, M.K.; Ree, B.; Cooper, J.; Jackson, J.; Young, J.; Lacewell, R.; Knutson, A. Economic impact of pecan integrated pest management implementation in Texas. *J. Econ. Entomol.* **1998**, *91* (5), 1011–1020.
11. Bumroongsook, S.; Harris, M.K. Distribution, conditioning and interspecific effects of blackmargined aphids and yellow pecan aphids (Homoptera: Aphididae) on pecan. *J. Econ. Entomol.* **1992**, *85* (1), 187–191.
12. Harris, M.K.; Li, T. The blackmargined aphid as a key-stone species: a predator attractor redressing natural enemy imbalances in pecan systems. In *Dynamics of Forest Herbivory*; USDA Forest Serv. Gen. Tech. Rep., 1996; NC-183, 112–117.
13. Harris, M.K.; Dean, D.A. Pecan foliar spray programs, patterns, and materials in Texas. In *Pecan Industry: Current Situation and Future Challenges*, Third National Pecan Workshop Proceedings, USDA-ARS: Ruidoso, New Mexico, 1998–2004, 1998; 40–54.
14. Harris, M.K.; Chung, C.S. Masting enhancement makes pecan nut casebearer pecans ally against pecan weevil. *J. Econ. Entomol.* **1998**, *91* (5), 1005–1010.
15. Harris, M.K. Pecan weevil management considerations. In *Pecan Industry: Current Situation and Future Challenges*; Third National Pecan Workshop Proceedings, USDA-ARS: Ruidoso, New Mexico, 1998–2004, 1998; 66–73.
16. Chung, C.S.; Harris, M.K.; Storey, J.B. Masting in pecans. *J. Amer. Soc. Hort. Sci.* **1995**, *120* (3), 386–393.
17. Harris, M.K. IPM, what has it delivered? A Texas case history emphasizing cotton, sorghum, and pecan. *Plant Dis.* **2001**, *85* (2), 112–121.
18. Harris, M.K. Integrated pest management of pecans. *Annu. Rev. Entomol.* **1983**, *28*, 291–318.
19. Thompson, T.E.; Grauke, L.J. Field resistance to yellow aphids in pecan. *J. Amer. Soc. Hort. Sci.* **1998**, *123* (1), 85–90.

Persistent Organic Pesticides

Agneta Sundén Byléhn

*UNEP Chemicals, United Nations Environment Program,
Chatelaine (Geneva), Switzerland*

INTRODUCTION

Persistent organic pesticides are part of a larger group of chemicals known as Persistent Organic Pollutants or POPs, which also includes industrial chemicals and unwanted by-products such as dioxins that are formed during incomplete combustion processes. Chemically, POPs include both polycyclic hydrocarbons and halogenated hydrocarbons, and all persist for long periods in the environment as they resist, to varying degrees, chemical, biological, and photochemical degradation.^[1] Furthermore, POPs bio-accumulate in fatty tissues, thanks to their lipophilic characteristics, and thereby bio-magnify through the food-chain causing adverse effects to health and the environment as concentrations build up in living organisms. During the last two decades much attention has been given to this group of substances at the international level after it became apparent that they are transported through the environment across borders. Several countries started banning these POPs in the 1970s; however, individual countries alone were unable to control the environmental pollution from such border crossing substances, and critical concentrations have been reached in some regions even in places where they have never been produced or used. A regional legal agreement that specifically addresses POPs was adopted in 1998 with the Aarhus Protocol on Persistent Organic Pollutants under the regional Convention on Long-Range Transboundary Air Pollution (LRTAP) of the UN Economic Commission for Europe (UNECE).^[2] However, a regional agreement was not enough and negotiations of a global legally binding instrument to reduce and/or eliminate releases of POPs were started under the auspices of UNEP in 1998. In May 2001, over 100 countries agreed and adopted this global treaty, now named the Stockholm Convention on Persistent Organic Pollutants.^[3] Several other international activities also address POPs, notably the Global Programme of Action for the Protection of the Marine Environment from Land-based Activities (GPA)^[4] and a number of regional seas agreements.

PERSISTENT ORGANIC PESTICIDES UNDER THE STOCKHOLM CONVENTION

There are presently 12 POPs covered by the Stockholm Convention of which nine are pesticides. The 12 substances are all chlorinated hydrocarbons and constitute an initial list of POPs, which can be expected to increase in the future since the Convention contains a procedure and criteria for adding new POPs as candidates for international action. (Table 1).

The Protocol on POPs under the LRTAP Convention covers the same nine pesticides as well as chloro-decane and hexachlorocyclohexane (HCH).

Parties can make proposals for adding new POPs to the Stockholm Convention once it has entered into force, which it does when 50 countries have ratified it. Proposals must contain information for both the chemical and its transformation products relating to the screening criteria that concern persistence, bio-accumulation, potential for long-range environmental transport, and adverse effects. A POPs Review Committee will review this and other information received from Parties, prepare a risk profile, make a risk management evaluation, and, based on these, it will make recommendations to the Conference of Parties which then decides whether to list the chemical in the Convention.

MAJOR ISSUES CONCERNING PERSISTENT ORGANIC PESTICIDES

The pesticides presently covered by the Stockholm Convention include many of the first generation of insecticides that after the second World War played an important role in combating vector-borne diseases and increasing food production. The use of these substances had remarkable effects, not the least DDT. When used for in-door residual spraying it saved millions of human lives in malaria eradication program during the 1950s and 1960s.^[5] Unfortunately, negative effects of these substances started to show up on the environment after some years, in particular on bird

Table 1 Initial list of pesticides covered by the Stockholm Convention

| |
|---|
| Aldrin |
| Camphchlor (toxaphene) |
| Chlordane |
| DDT |
| Dieldrin |
| Endrin |
| Heptachlor |
| Hexachlorobenzene (HCB) (also an industrial chemical and unwanted by-product) |
| Mirex |

populations, and in 1962 the public was alerted by Rachel Carson's *Silent Spring*, about the potential consequences of continued use of chlorinated pesticides.^[6] In more recent years, our understanding and concern about adverse effects from long-term low-level exposures to these persistent pesticides have greatly increased. Such potential long-term effects include cancer, suppression of the immune system, and reproductive, developmental, and neurological disturbances. Many of these are linked to the endocrine system which is affected by chemicals that mimic or block the functions of normal hormones, and during the last years there have been several reports describing such endocrine-related effects.^[7] Such effects cause great concern, in particular when they become apparent in more sensitive ecological systems such as the arctic, where the animals much more than elsewhere depend on their fatty tissues to survive. Unfortunately, semi-volatile POPs tend to get trapped in colder regions, thereby constituting a particular threat to ecosystems with cold climates including high mountainous areas. However, levels can also be significant in warmer climates as indicated in a report where residue levels in African fauna in 1995 were found to be significantly higher compared with levels in Europe and the U.S.A. in the 1970's when restrictions were initiated.^[8]

In addition to the negative impact on the environment and health, failures in controlling pests and vectors with these insecticides also became a problem. A first reaction was to increase insecticide amounts and treatments, but the problems continued. Eventually, the impacts of these pesticides on local ecological systems were better understood, and the reasons for the failures in crop protection became apparent, in particular the development of resistance to the insecticides. However, the ecological balances were also upset as the insecticides killed natural pest enemies, such as spiders, and secondary pests could gain ground and cause new problems.^[5]

PESTICIDES SCHEDULED FOR ELIMINATION AND RESTRICTION UNDER THE STOCKHOLM CONVENTION

All POPs pesticides except DDT have been scheduled for elimination under the Stockholm Convention, meaning that each party to the Convention shall eliminate their intentional production and use. There are, however, some uses for which it has proven difficult to immediately switch to alternative chemicals or other approaches.

DDT is scheduled for restriction, and its production and use for disease vector control are considered an acceptable purpose under the Convention. Parties can produce and/or use DDT for disease vector control when no locally safe, effective, and affordable alternatives are available, but production and use of DDT must be notified in a public DDT Register. Each party that uses DDT must provide information every 3 years on the amounts and conditions of use and the relevance of its in use in their disease management strategy. Parties will also be encouraged to develop an action plan inter alia to ensure that DDT use is restricted to disease vector control, and to implement suitable alternative products, methods, or strategies, including resistance management strategies, to ensure the continued effectiveness of such alternatives. The Conference of Parties will, in consultation with the World Health Organization, evaluate the continued need for DDT for disease vector control at least every 3 years. By May 2001, 32 countries had requested exemptions for use of DDT for disease vector control.

For other pesticides, except endrin and camphchlor (toxaphene), and for other uses of DDT, Parties may register for specific exemptions that are identified for each pesticide in Annexes A and B to the Convention. Termite control is the most apparent exemption, which is listed for three pesticides, namely, chlordane, heptachlor, and mirex. By May 2001, there were 11 countries that had requested such exemptions. Chlordane can also be used as an additive in plywood adhesive and heptachlor for wood treatment, and four countries have made such requests. There are also a few other exemptions that have been requested by only one country or that do not constitute a pesticide use.

In addition, the Convention also provides for general exemptions that apply to all intentionally produced POPs including exemptions concerning unintentional trace contaminants in products and articles and constituents of articles manufactured or already in use before or on the date of entry into force.

ALTERNATIVE APPROACHES TO POPs PESTICIDES

From the above, it is obvious that the major issues regarding the elimination of pesticides in the present list of POPs concern the use of DDT for disease vector control, in particular malaria mosquitoes, and the use of chlordane, heptachlor, and mirex for control of termites. In efforts to eliminate these, governments should seek alternative approaches that are sustainable. In particular, it will be important to ensure that these pesticides are not simply replaced by other pesticides, but that the principles of integrated pest and vector management are adopted. It will further be extremely important to ensure that the strategies used will not be compromised by measures in other sectors, as has happened when, for example, resistance has developed in disease vectors where the same or similar insecticides are used in agriculture, or environmental modifications have created breeding grounds for malaria mosquitoes. In its efforts to assist countries find more sustainable solutions to POPs, UNEP, jointly with World Health Organization and the Food and Agriculture Organization of the United Nations, is highly promoting close collaboration between sectors in order to identify and implement opportunities that can mutually benefit all sectors.^[9] Such solutions must be based on the local conditions and can be best sustained through active community participation. Structures established under one sector such as Farmer Field Schools may, for example, very well serve purposes of public health and the environment. The interrelationship among environment, agriculture, and health is, hence, a key

for identifying sustainable strategies that will effectively and efficiently protect agriculture from pests, communities from diseases like malaria, and ecosystems from persistent pesticides.

REFERENCES

1. Ritter, L.; Solomon, K.R.; Forget, J. *Persistent Organic Pollutants—An Assessment Report on: DDT-Aldrin-Dieldrin – Endrin – Chlordane – Heptachlor – Hexachlorobenzene – Mirex – Toxaphene – Polychlorinated Biphenyls–Dioxins and Furans*; Prepared for the International Programme on Chemical Safety (IPCS) within the framework of the Inter. Organization Programme for the Sound Management of Chemicals (IOMC), WHO: Geneva, 1995.
2. <http://www.unece.org/leginstr/cover.htm>.
3. <http://www.chem.unep.ch/pops>.
4. <http://gpanews.unep.org>.
5. UNEP/WHO/FAO Draft. *Reducing and/or Eliminating Persistent Organic Pesticides—Guidance on Strategies for Sustainable Pest and Vector Management*; United Nations Environment Programme: Geneva, in preparation.
6. Carson, R. *Silent Spring*; Houghton Mifflin: Boston, 1962.
7. <http://www.ourstolenfuture.org/>
8. Wiktelius, S.; Edwards, C.A. Organochlorine residues in African fauna. *Rev. Environ. Contam. Toxicol.* **1997**, *151*, 1–37.
9. UNEP/FAO/WHO. *Workshop on Sustainable Approaches for Pest and Vector Management and Opportunities for Collaboration in Replacing POPs Pesticides*; UNEP: Geneva, 2001.

Pest Eradication: Screwworm as a Model

Robert E. Reichard

Scientific and Technical Department, World Organization for Animal Health (OIE), Paris, France

INTRODUCTION

Pest eradication is a complicated undertaking requiring a well thought out integration of several aspects of pest management and even politics. The example of New World Screwworm in North and Central America provides a useful insight into how different program components, from the Sterile Insect Technique, the control of animal movement, surveillance and prophylaxis, fly trapping, and cooperation between nations, can combine to effectively control and eradicate a deadly pest.

PEST ERADICATION: SCREWWORM AS A MODEL

Screwworm myiasis, caused by infestation of wounds by the obligative parasitic larval stage of the New World Screwworm (NWS), *Cochliomyia hominivorax*, was, before its eradication from the southern United States, a major disease constraint to livestock production there. During periods of high infestation in tropical and subtropical America, morbidity is high, and untreated wounds often result in mortality. The prototype and still most successful application of the Sterile Insect Technique (SIT) was developed by Knippling in the 1950s for NWS. Programs beginning in Florida have continued to date to eradicate NWS from North America southward to Panama, where a sterile fly barrier to protect the continent will be established (see Fig. 1). Although screwworms have been eradicated only with the application of SIT, this technique alone will not eradicate the pest. Other program elements are required to free an area and maintain it free of NWS.

THE DISEASE

Although, as far as is known, all warm-blooded creatures including humans can be infested, the parasite's major impact is on livestock. Even minor wounds such as the umbilicus of newborns, scratches, or tickbites offer living flesh required for nutrition of larvae resulting from laying of more than 300 eggs on their edges

by each female fly. Enlarged wounds then attract further infestation. The major cost to livestock owners is continuous surveillance for essential prophylactic treatment of wounds. Before eradication began in the southwest U.S. in 1960, the costs of infestation there were estimated at \$100 M annually.^[1] Later, much larger estimates reflect not only inflated dollars, but also the lack of available cowboys whose major responsibility in the 20th century was to ride the range to prevent screwworms. Small and part-time breeders also profited and now operate on the basis of NWS absence.^[2]

STERILE INSECT TECHNIQUE

The development of the SIT has been extensively documented, as has its application for NWS eradication.^[3] Mass production of hydroponically grown and radioactively sterilized NWS—500 million/week was required for aerial dispersion during the height of eradication in Mexico during the early 1980s—is applied entomology on an industrial scale, requiring biologically sensitive systems for all phases including delivery to often distant dispersion sites. Sterile pupae in Mexico were transported in refrigerated trailers up to 2400 km to packaging sites, where they were redistributed in boxes at eclosion temperatures to airfields for immediate dispersion in twice weekly grid patterns until eradication occurred. In an outbreak in Libya, an emergency program under United Nations auspices was set up to eradicate a 20,000-km² focus originating from imported animals before it could spread throughout the Mediterranean basin and beyond. Forty million sterile flies from the world's only plant for the purpose in southern Mexico were required weekly with considerable development of methods needed to transport pupae in viable condition until eclosion and delivery to dispersion sites.^[4]

Development of vigorous sterile NWS production strains and their periodic changing as they degenerate over generations due to less than ideal rearing conditions or simple "factory domestication" is the most critical element for SIT success. Genetic selection, breeding, testing, and adaptation to artificial rearing is a continuous process normally exceeding 6 mo



Fig. 1 New World Screwworm eradication progress in North America. (Courtesy of the Office International des Epizooties.)

before a new mass production strain is ready. Unpredictable, possibly rapid failures of the ongoing strain require backup strains with the biosecurity this entails. There have been periods in the 40 yr of programs when less than ideal strains for SIT slowed eradication progress.^[5]

Biosecurity in a plant producing hundreds of millions of NWS weekly is a challenge. Although backup security systems ensure that only irradiated pupae leave the plant for dispersion, other precautions so that highly mobile adults do not escape the plant are needed. Sterile fly dispersion in the vicinity is only a partial answer to this continuous problem.

ANIMAL MOVEMENT CONTROL

Aerial dispersal of adult NWS flies is an important part of their natural history, and waves of generations from endemic areas of the southern United States and Mexico seasonally accounted for cases far into temperate zones, where they were unable however to overwinter (mean daily temperature less than 10°C for consecutive months).^[6] During the 1960s and 1970s when conditions were suitable, NWS from Mexico breached a 3000-km-long sterile fly barrier deployed along both sides of the border between the two countries.

The transport of infested animals, however, is by far the most important reason for the long distance spread of screwworms. Larval maturation inside wounds is

about a week, and the location of an affected animal when they fall to the ground to pupate, if conditions are appropriate, determines the site of a future outbreak. Throughout the programs, screwworms have been trucked in animal wounds through large curtains of sterile flies to reinfest areas that had been cleared of the parasites. This occurred in Mexico until adequate quarantine stations with Army enforcement ensured that all animals transiting a southern barrier northward were individually examined, wounds treated, and prophylactically dipped.

The 2 yr between confirmation of the presence of NWS in Libya in January 1989 until the availability of sterile flies from the plant in Mexico amply demonstrated the value of effective animal movement control. Fixed and mobile quarantine stations were established at the periphery of the infested area. Although the intensity of infestation within the area increased dramatically, it had expanded from 20,000 km² to only 25,000 km² by the time dispersion, which quickly resulted in eradication, began.^[5]

Since eradication NWS continues to be reintroduced into the United States and Mexico (and some other free countries) with infested animals, and occasionally people. Fortunately, all cases to date have been suppressed with SIT, have been detected in quarantine or clinics, or have occurred under conditions where there was no reproduction.

SURVEILLANCE AND PROPHYLAXIS

In the Americas, NWS programs have traditionally combined surveillance and prophylactic treatment of wounds in the form of widespread provision of treatment and larval sampling kits to animal owners before, during, and subsequent to eradication of an area where SIT was employed. Owners were continuously urged to treat wounds using individual residual insecticide packets and obtain larvae if present from the deepest parts of wounds so as not to sample only non-malignant blowflies, which frequently accompany NWS infestation, resulting in a false negative diagnosis. Samples were submitted to program personnel for identification. Because treatment of infested and noninfested wounds alike was essential and the benefits of eradication to livestock owners was very evident, cooperation was usually good.

Intensive public relations promoted this sampling, which was used to continuously define both the geographic limits and intensity of NWS infestation. By measuring the ratio of positive to negative samples, the suppression of NWS by SIT was revealed, as was the degree of cooperation by livestock raisers. If few blowfly samples from wounds were submitted during the season, efforts to increase surveillance in that area

were necessary. A regular flow of NWS-negative fly larvae samples after SIT was completed helped assure program officials that eradication had indeed been accomplished.^[2]

FLY TRAPPING

Adult NWS field sampling with a series of traps baited with an attractant mimicking the odor of wounds, primarily to attract female flies, is very labor-intensive and employed normally to measure critical aspects of SIT. A large number of NWS females are dissected daily to determine their fertility. The ratio of sterile to fertile trapped females indicates progress in an area under SIT treatment. The vigor and flight range of a new sterile fly production strain or its condition later on can be measured. Eradication problem areas may also be investigated with trapping.

Because wound sampling by livestock owners can be practical and successful in surveying large areas, trapping was used infrequently for the purpose until the outbreak in Libya, during which a large number of people in a relatively small area were trained in the techniques. Although parallel larval sampling and fly trapping programs defined as expected the same geographic limits of infestation, more fertile NWS flies than larvae were detected during SIT, demonstrating that trapping can be a sensitive surveillance technique.^[4]

INTERNATIONAL COOPERATION

Screwworm eradication was not only the prototype for the successful use of SIT but also a dramatic example of international cooperation by governments and livestock raisers alike, to rid as of this writing almost an entire continent of an important animal pest. The surveillance so essential for eradication was primarily the responsibility of livestock owners. Southwestern U.S. livestock raisers provided the seed money to begin SIT and pressured their government to establish with their cooperation an eradication program. When Mexican producers saw results of a barrier across their northern states, they joined their American colleagues to influence both governments to form a Mexico

United States Commission to eradicate NWS to the Isthmus of Tehuantepec in southern Mexico. As barriers require costly continuous sterile fly dispersion, the even narrower and remote Isthmus of Darien in southern Panama provided an incentive for Central Americans to join to proceed southward where NWS will be more economically contained.

A perhaps even more remarkable example of international cooperation was eradication of an exotic NWS outbreak from Libya by that government under UN auspices in partnership with several countries, including some political adversaries such as the United States, in order to prevent the pest from becoming endemic in the Eastern Hemisphere.

Since the late 1950s, U.S. resources alone devoted to NWS eradication exceed \$750 million. Contributions of Latin American neighbors and particularly Mexico, where eradication efforts were of a very large scale, bring the total expenses to nearly \$1 billion to achieve this ambitious goal. Livestock breeders throughout the continent, who have contributed considerable additional resources to the effort, are unlikely to let their governments allow any reversals so that they would have to live again with the pest.

REFERENCES

1. Novy, J.E. Screwworm control and eradication in the southern United States of America. *FAO World Anim. Rev.* **1991**, 18–27, Special Issue: October.
2. Reichard, R.E. New World Screwworm and its appearance in the Eastern Hemisphere. *FAO World Anim. Rev.* **1991**, 1, 40–47.
3. Graham, O.M., Ed.; *Symposium on the Eradication of the Screwworm from the United States and Mexico*; Miscellaneous Publication No. 62; Entomological Society of America: Hyattsville, 1985.
4. Food and Agriculture Organization of the United Nations (FAO). *The New World Screwworm Eradication Program: North Africa 1988–1992*; FAO: Rome, 1992.
5. Reichard, R.E. Case studies of emergency management of screwworms. *Rev. Sci. Tech. Off. Int. Epizoot.* **1999**, 18 (1), 145–163.
6. Krafur, E.S.; Lindquist, D.A. Did the sterile insect technique or weather eradicate screwworms (Diptera: Calliphoridae) from Libya? *J. Med. Entomol.* **1996**, 33 (6), 877–887.

Pest Tolerance in Crops

Vicki Tolmay

*Small Grain Institute, Agricultural Research Council,
Bethlehem, South Africa*

INTRODUCTION

Crops tolerant to pests and diseases are able to produce higher yields or a better-quality end product than would have been produced by a susceptible crop without the use of pesticides. Tolerance to pests and diseases has been used for many years. In the late 19th century, grape phylloxera all but destroyed the French wine industry but, by grafting resistant American rootstocks to French scions, the industry recovered.^[1] In the early 1900s, potato cultivars resistant to late blight were introduced following the disastrous epidemic in Western Europe in the 1840s, which led to widespread famine.^[2] Natural selection of more tolerant genotypes, as well as intentional improvement through research and development, has been taking place for many years; it is now safe to say that almost all crops cultivated today possess a certain level of tolerance to some pest.

BACKGROUND INFORMATION

In a tolerant crop, the level of pest control ranges from adequate to good, but is usually far from perfect. Benefits such as reduced pesticide use and the positive impact this has on the environment and human health are perceived as the direct value of this form of pest management. Although research into crop tolerance has been undertaken on a wide variety of crops for decades, there are as many failures as successes.^[1–3] Unfortunately, other factors besides the efficacy of the pest control collude to prevent tolerant crops from being utilized to their full potential. Pest tolerant crops are only adopted and cultivated by farmers when other factors such as yield, agronomic characteristics, appearance, processing characteristics, and taste are acceptable to the producers and end users. Tolerant genotypes of many crop species are known to science and probably many more are still to be discovered, but they will need to be incorporated into acceptable cultivars before this tolerance can show any impact. The cost of developing tolerant crops can be a problem; the improvement of less important crops is often not possible due to a lack of funding rather than a lack

of suitable germplasm and scientific techniques.^[1] Numerous accessions of various crops are maintained in germplasm banks around the world^[1,2,4,5] and could be used in future research efforts.

TOLERANT CROPS IN THE 21ST CENTURY

If pest tolerance in crops has been known for so long and has decided advantages over other control methods, why has it not been utilized more? No simple explanation can be given but factors such as the long period of time needed to breed better cultivars, particularly in crops like fruit trees, and the cost of such breeding undoubtedly played a role. Combining tolerance to different pests in a multiple pest tolerant crop has also been difficult and may be easier in future using breeding techniques like genetic engineering. The most important reason, however, is related to the people using the tolerant crop—its producer, processor, and end user. The impact of pest tolerance in crops is a question of relativity and boils down to a single issue: What level of pest damage to the crop is tolerable to the end user? Very broadly speaking, there are two major groups in which pest tolerant crops can be classified.

Crop Tolerance in Staple Foods

Crops such as rice, cassava, maize, sorghum, wheat, and grain legumes including dried beans, chickpeas, and lentils, to name but a few, form the staple food of billions of people in the developing world. For subsistence and/or developing communities, which are motivated by the need to prevent pests from diminishing their primary food source, the social and economic impacts of a tolerant crop are profound—a matter of life or death in some cases. Crop tolerance is very well suited for use in resource-poor circumstances as it functions independently of the socioeconomic constraints of the producer. It is not influenced by factors such as literacy, cash flow, education level, pesticide application principles, and safety requirements. The Consultative Group on International Agricultural Research (CGIAR) has used pest tolerant crops to

great effect by reducing poverty and ensuring sustainable food security through its many International Research Centers. More than 100 rice cultivars with resistance to insect pests have been developed at the International Rice Research Institute^[1,5] Wheat germplasm coming from CIMMYT (International Maize and Wheat Improvement Center) contains durable resistance to a number of important diseases, notably stem and leaf rust.^[5] More than 180 improved bean varieties with tolerance to diseases and pests, bred from International Center for Tropical Agriculture (CIAT) germplasm have been released and are widely planted.^[5] These tolerant crops play an influential role in stabilizing production levels in many regions affected by mass poverty, hunger, and malnutrition and contribute immensely to global food security. Despite these efforts, famine has not been eradicated and in the final equation, it seems that the benefits of tolerant crops are often negated by market, policy, institutional, and organizational failures.^[6]

Crop Tolerance in Non-Staple Foods

The cosmetic appearance of food is an important component of the production and marketing processes in many nonstaple foods. Tolerant crops by definition do not necessarily eradicate the pest and therefore physical evidence of the presence of pests is often visible on the crop itself in the form of marks and scars. Some crops in which tolerance to pests and diseases has been used are apple, coffee, tomato, potato, sugar beet, strawberry, cotton, pear, sugarcane, lettuce, tobacco, cocoa, raspberry, okra, muskmelon, and banana.^[1,2] Varying levels of success have been achieved. The smaller impact of crop tolerance to pests in nonstaple foods recorded to date, particularly fruit and vegetables, is influenced by an intricate play of sociological and emotional factors. The food industry, public opinion, and preference, in combination with buying ability, determine the acceptable cosmetic standards of nonstaple food. The bottom line is that most consumers are persuaded to buy good-looking food. Ideologically, if people could be persuaded that an ecologically sound production practice such as crop tolerance to pests produces better food, albeit not as aesthetically pleasing, they might be persuaded to buy it. At present, a very small proportion of the world population buys its food from specialty shops and organic markets, paying a premium for more "healthy" food. Differences in socioeconomic circumstances and perceptions of scientific intervention in crop production determine the consumers' attitude toward the technology involved in the production of the food they buy. Though not impossible, it will be difficult to change established consumer patterns.

CAN CROP TOLERANCE REDUCE PESTICIDE USE?

This depends entirely on the particular crop, its pest complex, and production circumstance. Pesticide use can be reduced by using tolerant crops, as the Russian wheat aphid resistant wheat cultivars in South Africa showed. Adoption of these cultivars by farmers was rapid and led to a dramatic reduction in insecticide use in the eastern Free State Province. The percentage of farmers using insecticide sprays decreased by 43% from 1990 to 1997, and a further decrease of 16% was projected by 2000. The average area treated with insecticides decreased from 85% in 1990 to 30% in 1997 and was projected to decrease to only 16% in 2000. The number of sprays per year decreased from four times during 1990–1992 to only one time after 1996.^[7] Crop tolerance and pesticides can be used in conjunction to obtain better results.^[1,8,9] In Brazil, the combination of fungicide seed treatment and slow leaf and panicle blasting resistance gave a significant increase in rice yield,^[9] and in the United States, sorghum midge resistant hybrids responded more efficiently to insecticide treatment.^[1] The reverse is also true in that some insects reared on resistant plants show increased tolerance to insecticides,^[8] making them even more difficult to control. Sometimes using tolerant crops reduces pesticide treatment of the major pest, but minor pests, to which the crop is not tolerant, resurge and can only be controlled by spraying—the end result is no reduction of pesticides.

The big dilemma is that crop tolerance can be an economically viable pest control option and can impact positively on human health and welfare; but this does not happen to as large an extent as it could. This is probably due to the fact that its successful deployment is influenced by so many other factors. Pest resistant crops have many potential benefits, but these can only be realized on a large scale if people are committed to making these crops work effectively.

REFERENCES

1. Smith, C.M. *Plant Resistance to Insects: A Fundamental Approach*; Wiley: New York, 1989; 1–285.
2. Russell, G.E. *Plant Breeding for Pest and Disease Resistance*; Butterworth: London, 1981; 1–485.
3. Shanower, T.G.; Romeis, J.; Minja, E.M. Insect pests of pigeonpea and their management. *Annu. Rev. Entomol.* **1999**, *44*, 77–96.
4. Bellotti, A.C.; Smith, L.; Lapointe, S.L. Recent advances in cassava pest management. *Annu. Rev. Entomol.* **1999**, *44*, 343–370.
5. Impact of the CGIAR. <http://www.cgiar.org/impact.htm> (accessed April 2000).

6. Von Braun, J.; Msuya, J.; Wolf, S. On the 'How to' of agricultural growth promotion and improved food security: implications for Southern Africa in a regional and international context. In *Challenges Facing Agriculture in Southern Africa, Agrekon Special Issue, Vol. 38*, Interconference Symposium of the International Association of Agricultural Economists, Badplaas, South Africa, August 10–16, 1998; Kirsten, J., Van Zyl, J., Hassan, R., Eds.; Agricultural Economics Association of South Africa: Silverton, 1999; 1–19.
7. Marasas, C. Wheat management practices and adoption of the Russian wheat aphid integrated control program in the study area. In *Socio-Economic Impact of the Russian Wheat Aphid Integrated Control Program*; Ph.D. thesis in the Department of Agricultural Economics, Extension and Rural Development, Faculty of Biological and Agricultural Science, University of Pretoria: Pretoria, South Africa, 1999; 121–183.
8. Van Emden, H.F. The role of host plant resistance in insect pest mismanagement. *Bull. Entomol. Res.* **1991**, *81*, 123–126.
9. Filippi, M.C.; Prabhu, A.S. Integrated effect of host plant resistance and fungicidal treatment on rice blast control in Brazil. *Plant Dis.* **1997**, *81*, 351–355.

Pest-Free Planting Stock: Selection

V. J. Shivankar

Department of Entomology, National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

Shyam Singh

National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

INTRODUCTION

The term “pest-free” stock indicates the absence of pests and pathogens on or in the propagation stock. Mere absence of symptoms of disease or a malady and its effects may not really represent the “pest-free stock” because various environmental conditions, cultural practices, or tolerance of the cultivar may mask visual disease symptoms. Most of the vegetatively propagated crop plants are systemically infected with one or more pests/pathogens such as insects, mites, nematodes, and disease-causing pathogens ranging from fungi to viroids drastically affecting yield and quality of produce. In the absence of an organized budwood certification program, nurseries inadvertently multiply and supply infected planting materials. Cultivation of such plants is a major cause of decline in citrus.^[1]

SELECTION OF MOTHER PLANTS

Mother plants that attained maturity with exceptionally good health, vigor, size, and record of consistently high yield of quality produce at least for 5 years, free from systemic virus diseases, are selected through extensive surveys,^[2] and are kept under protected cover (insect proof). Thus, pedigree-selected commercial source orchards or vineyards may be used to provide buds, scions, or cuttings or fruit, nut, and vine crops. To obtain planting stock of vegetative propagating crops, it is essential that they meet the following attributes: 1) true-to-name and type; 2) free of diseases and insect pests; and 3) proper physiological state so that the grafts, buds, and cuttings taken from them will take root properly.^[3]

To produce “disease-free” plants, a healthy nucleus stock could be developed by selecting out one or more healthy plants and then multiplying them vegetatively, but where the entire population of a clone is infected, the only way to obtain a “pathogen-free plant” is through tissue culture. Using the apical portion of vegetative shoots and discarding lower portions can often avert the possibility of selecting tissues infected with organisms that cause vascular wilt, e.g. *Fusarium*, *Verticillium* and *Phytophthora*. It has become increasingly

common to use tissue culture-produced liners as sources of stock plants in the development of new cultivars and disease-indexed plants. Conventional macropropagation techniques can then be used after stock plant establishment (Table 1).

SOURCE SELECTION

Large genetic advances can be made in a single step by selecting a single unique superior plant from a seedling population and reproducing it asexually by vegetative propagation. Exploitation of apomixis should play a key role in the case of sexually propagated crops. This may occur when propagating minor commercial cultivars, establishing collections, transporting through quarantine barriers, or beginning a nuclear stock program. Because such a plant becomes the sole representative of that clone in future propagation, it constitutes a new source clone. Propagation then takes place in a sequential pattern in both time (vertical) and space (horizontal), and provides an historical vegetative pedigree for the cultivar.^[4]

Nuclear Stock Selection

This program include the following steps:

1. Initial selection of a nucleus of individual source plant(s). Candidate plants are tested for genetic potential;
2. Maintenance of nuclear stock in special blocks with safeguards against reinfection and genetic change; and,
3. A system of commercial propagation and distribution whereby source material is multiplied and disseminated without reinfection and/or genetic change.

DETECTION OF PATHOGENS

Culture Indexing

The principle of “culture indexing” is to place pieces of plant tissues in aseptic culture, via a medium favoring

Table 1 Some plant species for which pathogen free plants have been obtained by tissue culture techniques^a

| Plant species | Virus bacteria/fungi eliminated |
|--|---|
| <i>Allium sativum</i> (garlic) | Garlic mosaic virus, onion yellow dwarf mosaic virus, garlic yellow streak virus |
| <i>Ananas sativus</i> (pineapple) | Unspecified |
| Begonia (hybrids) | Bacterium: <i>Xanthomonas begoniae</i> |
| <i>Brassica oleracea</i> (cauliflower) | Cabbage black ringspot virus, turnip mosaic virus, cauliflower mosaic virus |
| Carrot (MLOs) | Aster diseases |
| <i>Chrysanthemum</i> sp. | Chlorotic mottle, complex of viruses, green flower, stunt, tomato aspermy, vein mottle, virus B |
| <i>Dahlia</i> spp. | Complex of viruses, dahlia mosaic, tomato aspermy, vein mottle, virus B |
| <i>D. caryophyllus</i> (carnation) | Complex of viruses, etched ring, latent, mottle, streak, ringspot, unidentified, vein mottle Bacteria: <i>Pseudomonas carophylli</i> and <i>Pectobacterium parthenii</i> |
| <i>Forsythia</i> × <i>intermedia</i> | Complex of viruses, crinkle, edge, latent A, |
| <i>Fragaria</i> sp. (strawberry) | latent c, mottle, pallidosis, strawberry, yellow edge, vein banding, yellow virus complex, vein chlorosis |
| <i>Gladiolus</i> spp. | Unidentified viruses Fungi: <i>Fusarium roseum</i> , <i>Fusarium cerialis</i> , <i>Verticillium</i> |
| <i>Glycine max</i> (soybean) | Soybean mosaic virus |
| <i>Hydrangea macrophylla</i> | Hydrangea ringspot, |
| <i>Impomoea batatas</i> (sweet potato) | Feathery mottle hanmon mosaic, internal cork, rugosa mosaic, synkuyo mosaic, unidentified |
| <i>Lilium</i> spp. | Cucumber mosaic virus, MyMV, lily, symptomless, latent, lily mosaic virus, unidentified |
| <i>Lolium multiflorum</i> (ryegrass) | Ryegrass mosaic virus |
| <i>Malus</i> sp. (apple) | Latent viruses |
| <i>Malus pumila</i> (apple) | Apple chlorotic leafspot virus |
| <i>Manihot</i> sp. (cassava) | African cassava, mosaic, cassava brown streak mosaic, unidentified |
| <i>Musa</i> sp. (banana) | Cucumber mosaic virus, unidentified |
| <i>Musa acuminata</i> × <i>Musa balbisiana</i> | Musa mosaic virus |
| <i>Nicotiana tabacum</i> | Dark-green islands of tobacco mosaic virus |
| <i>Ornithogalum</i> | Ornithogalum mosaic virus |
| <i>Pelargonium</i> sp. | Cucumber mosaic virus, tomato black ringspot, tomato ringspot, unidentified |
| <i>Petunia</i> sp. | Tobacco mosaic virus, tobacco necrosis |
| <i>Pelargonium</i> | Bacterium: <i>Xanthomonas pelarganii</i> |
| <i>Polyanthes tuberosa</i> | Mosaic |
| <i>Ranunculus asiaticus</i> | Unidentified |
| <i>Rheum rhaponticum</i> (rhubarb) | Tobacco rattle, cucumber mosaic virus, cherry leaf roll virus, strawberry latent, ringspot, turnip mosaic virus |
| <i>Ribes grossularia</i> (gooseberry) | Vein banding |
| <i>Rubus ideaus</i> (raspberry) | Mosaic |
| <i>Saccharum officinarum</i> (sugarcane) | Mosaic |
| <i>Solanum melongena</i> (eggplant) | Eggplant mottled crinkle virus |
| <i>Solanum tuberosum</i> (potato) | Leaf roll, paracrinkle, potato aucuba mosaic virus, potato spindle tuber viroid, PVA, PVG, PVM, PVS, PVX, PYY |
| <i>Vitis vinifera</i> (grapevine) | Grapevine fanleaf virus |
| <i>Zingiber officinale</i> (ginger) | Mosaic |

^aThe main sources of information provided in this table are in Refs.^[10-12].
(From Ref.^[4].)

the growth of the pathogen for subsequent detection, identification, and characterization.

Virus Indexing

Virus indexing is a process of testing plants for the presence or absence of viruses. Every meristem tip or callus-derived plant must be tested before using it as a mother plant to produce “virus-free stock.” Many viruses have a delayed resurgence period in cultured plants. This necessitates indexing of plants at periodic intervals by such methods as sap transmission test, serology, and EM examination.

Biodiagnosis

The primary method of detecting viruses is to transmit them by grafting or budding to a sensitive indicator plant under insect-proof, controlled conditions (biological indexing), which then develops identifiable symptoms within a certain length of time. Certain viruses can be detected in herbaceous hosts by mechanical transfer of sap. Thus, virus-free plants among selected elite mother plants are identified for further multiplication.

Serodiagnosis

This test is performed by adding a drop of centrifuged sap from a test plant to a drop of antiserum taken from the blood of a rabbit. If the virus is present, the precipitation will take place because of the presence of specific antibodies in the blood. Serology identifies unique proteins associated with particular pathogens. It can also be combined with immunosorbent electron microscopy. Enzyme-linked immunosorbent assay (ELISA) is one of the serological methods used to identify virus(es) based on antibody (monoclonal and polyclonal) reaction, viz., citrus tristeza virus, mosaic, ringspot, exocortis, polyviruses, greening bacterium, etc. ELISA is not applicable to viroids and viruses which have lost their coat proteins, and in diseases that involve several related luteoviruses such as potato leaf roll virus. All viruses may not react with the antiserum prepared against the main virus. Such agents can be detected through cDNA/cRNA probes or by RT-PCR assays.

ELIMINATION OF PATHOGENS FROM PLANTING STOCK

Heat Treatment (Thermotherapy)

At temperatures higher than optimum, many viruses in plant tissues are partially or completely inactivated with little or no injury to the host tissues.

Chemical Treatment

The use of virazole and vidarabine (antimetabolites) in the culture medium have resulted in virus-free lily and apple plants production.^[5]

Meristem Tip Culture

Excision and aseptic culture of the small pathogen-free apical dome of a growing segment can be the start of “clean nuclear stock.”

Micrografting

The meristems are grafted onto a virus-free rootstock (seedling) maintained and propagated in vitro.

Somatic cell hybridization, gene transformation, and somaclonal variation are other in vitro methods that can be utilized for regeneration of plants for disease resistance.

MAINTENANCE AND PREVENTION OF REINFECTION OF PEST FREE STOCKS

“Pest-free stocks” are maintained in sterilized soils in a glasshouse or insect-proof cages. Large-scale multiplication of these plants can be carried out by growing them in fields in isolated areas where chances of reinfection are minimal or none at all. The planting is usually referred to as a foundation (or mother) block. A limited amount of foundation propagating material is provided from a foundation (or mother) block. It is then multiplied to provide a nursery source block sufficiently large to provide propagules for commercial propagation. Alternatively, meristem-tip derived, virus-tested plants can be multiplied and maintained more easily and cheaply in cultures. Such plants, in general, have no additional resistance to diseases and may become quickly reinfected if proper precautionary measures are not adopted in the greenhouses, e.g., 1) control of disease vectors such as insects and nematodes by continuous spraying; 2) strictly enforced hygiene in a greenhouse. Pest control by sanitation is accomplished mainly by physical and chemical methods, including sterilization of the surface of seeds or tools by heat or chemical-disinfectants, physical separation of disease-free tissue from infected plants. Biological control introduced on top of good sanitation can extend the period of pest and disease control; 3) germ-free pots and substrates; 4) if proper precautionary measures cannot be maintained, the disease-free plant material can be maintained in vitro; 5) individual plants, both test or indicator, should be separated so as not to allow

them any contact with each other; and 6) indexing of each plant for diseases to be carried out at periodic intervals.

REGULATORY CONTROL

Because of the potential risk of transporting dangerous pests on the material or systemically infected clonal material, e.g., viruses, specific regulations and sometimes quarantines are in place to control such movement.^[6] Soil on the plant is not usually allowed to preclude the possible introduction of nematodes and other pests. It is important that propagators be familiar with national, state, and local regulations affecting the distribution of their products. At the national level, it would be of utmost importance to have regulated movement of planting material through a system of internal quarantine. There is a need to establish an appropriate budwood certification program.

COST-BENEFIT ANALYSIS

The selection of “pest-free planting stock” opens up a number of opportunities for cost minimization and profit maximization, irrespective of the crop, through realization of higher quality output. It has a definite comparative advantage from others, when all the underlying risks and uncertainties are taken into account.

The process of “selection and production of pest-free stocks” depends on technology access, material inputs like propagules, fertilizers, pesticides, chemicals, etc., availability of trained manpower, and presence of infra-structural facilities. Monitoring and surveillance of the planting stock for pests at both nucleus and foundation blocks, and also postrelease management of such materials are the major areas involving high cost (Fig. 1).

Yield increases of up to 300% (averaging 30%) have been reported following replacement of virus-infected stock with specific pathogen-free plants.^[7] In rhubarb, the petiole yield increased by 60–90% as a consequence of virus eradication.^[8] Eradication of viruses and other pathogens is thus highly desirable to optimize the yield and also to facilitate the movement of living plant materials across international boundaries.^[9] Commercialization of “disease-free planting material” in many crops like banana, citrus, strawberry, potato, etc., are the best known examples that strengthen the global food security by many folds.

Recent intensification of plant movements have resulted in increasing contamination by several debilitating diseases. This has resulted in a poor economic return on investments, while conversely “disease-free planting material” carefully established in a protected environment shall sustain a long-lasting and profitable crop production. The initial costs involved are high, but once the infrastructure is developed, economic feasibility studies show that production of the planting stock becomes cheaper in the long run through optimization

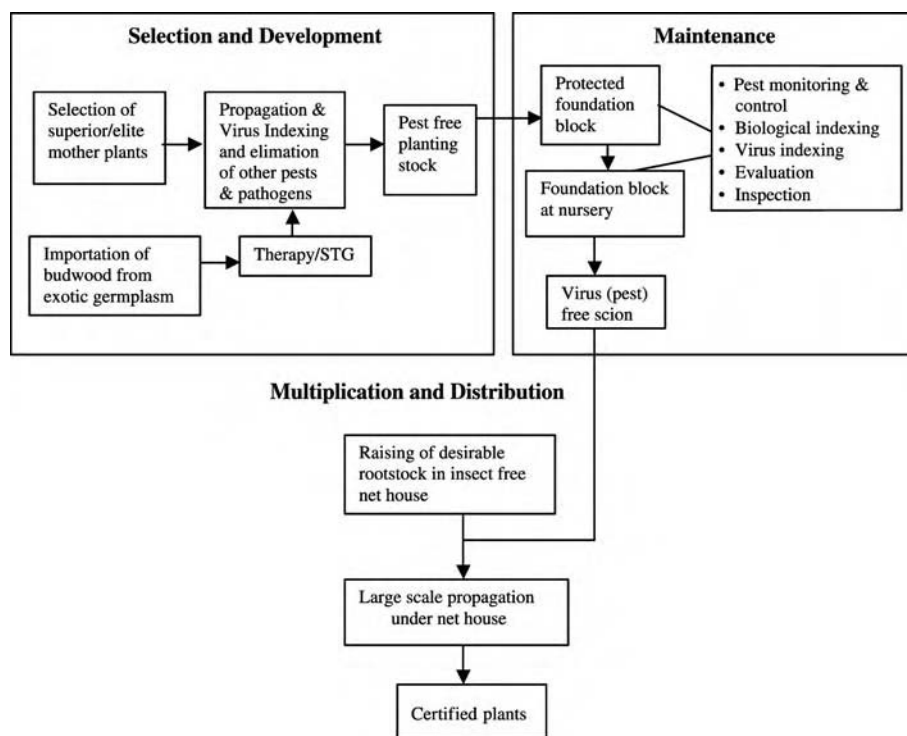


Fig. 1. Development of pest free planting stock. Includes: visual inspection of source (phenotypic selection); indexing (as required at each step); visual inspection of progeny (genotypic selection).

of resources and maximization of profits ensuring sustainable benefits both to consumers and producers.

REFERENCES

1. Shyam, S.; Shivankar, V.J.; Naqvi, S.A.M.H.; Singh, I.P.; Ghosh, D.K.; Das, A.K. *Production of Disease Free Planting Material of Citrus in India*, A Research Paper Accepted for Presentation in 6th International Congress of Citrus Nursery Management, Ribeirao Preto, SP, Brazil, July 9–13, 2001.
2. Zeigler, L.W.; Wolfe, H.S. *Citrus Growing in Florida*; University of Florida Press: Gainesville, FL, 1961.
3. Hudson, T.H.; Dale, E.K.; Fred, T.D. Source selection in clonally propagated cultivars, ctock plants; sources of cutting material and stock plant manipulation. In *Plant Propagation*; Prentice Hall International, Inc.: Englewood Cliff, NJ, 1990; 189–190, 271–272.
4. Bhojwani, S.S.; Razdan, M.K. Propogation of pathogen-free plants. In *Plant Tissue Culture: Theory and Practice, a Revised Edition*; Elsevier: Amsterdam, 1996; 451–482.
5. Pierik, R.L.M. *In Vitro Culture of Higher Plants*; Martinus Nijhoff Publishers: Dordrecht, The Netherlands, 1989.
6. Foster, J.A. Regulatory actions to exclude pests during the international exchange of plant germplasm. *Hort Science* **1988**, *23* (1), 60–66.
7. Murashige, T. Plant growth substances in commercial uses of tissue culture. In *Plant Growth Substances*; Skoog, F., Ed.; Springer: Berlin, 1980; 426–434.
8. Walkey, D.G.A.; Cooper, V.C. Comparative studies on the growth of healthy and virus infected rhubarb. *J. Hortic. Sci.* **1972b**, *47*, 37–41.
9. Button, J. International exchange of disease-free citrus clones by means of tissue culture. *Outlook Agric.* **1977**, *9*, 155–159.
10. Quak, F. Meristem culture and virus-free plants. In *Applied and Fundamental Aspects of Plant Cell, Tissue, and Organ Culture*; Reinert, J., Bajaj, Y.P.S., Eds.; Springer Verlag: Berlin, 1977; 598–615.
11. Walkey, D.G.A. In vitro methods for virus elimination. In *Frontiers in Plant Tissue Culture*; Thorpe, T.A., Ed.; University Calgary Press: Calgary, Canada, 1978; 245–254.
12. Wang, P.J.; Hu, N.Y. Regeneration of virus-free plants through in vitro culture. In *Advances in Biochemical Engineering: Plant Cell Culture II*; Fiechter, A., Ed.; Springer-Verlag: Berlin, 1980; 61–99.

Pesticide Labels

William Smith

Department of Entomology, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

Pesticides are toxic products that are intended to prevent, destroy, repel, or mitigate pests (such as insects, plant diseases, and noxious weeds), or are used as a plant growth regulators, defoliants, or desiccants. Pesticides are regulated in the United States by the Environmental Protection Agency (EPA), primarily through two federal statutes: the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA) and the Federal Food, Drug, and Cosmetic Act (FFDCA). Pesticide products must be initially registered by the EPA and then by individual states prior to distribution, sale, or use. A label, developed by a pesticide manufacturer/registrant and approved by the EPA, is required to be placed on all pesticide containers prior to the distribution, sale, or use of the product by the public.

A pesticide label can mean different things to different people. To a manufacturer/registrant, a label is a “license” for product distribution, sale, and use; to an enforcement official, a label represents the legal sale, use, and disposal of a product. A label provides the user with directions for correct and legal use to control a pest problem and, finally, the label provides valuable information to a physician in case of a pesticide poisoning accident.

LABEL VS. LABELING

The term “label” means the written, printed, or graphic matter on, or attached to, the pesticide product or device or any of its containers or wrappers.^[1] The term “labeling” means:

- All labels and all other written, printed, or graphic matter accompanying the pesticide product or device at any time.
- Reference made on the label or in literature accompanying the pesticide or device—an exception to this would be where reference is made in official publications by the EPA, U.S. Department of Agriculture (USDA), colleges, universities, state experiment stations, or agencies authorized by law to conduct pesticide research.
- Material Safety Data Sheets (MSDS) that come under the authority of the Occupational Safety

and Health Administration (OSHA) and are not reviewed by EPA—for the most part they are not considered as pesticide labeling. However, in certain cases, such as when an MSDS is intentionally distributed with a pesticide product, it then becomes part of the labeling.^[1]

LABEL DEVELOPMENT

The FIFRA requires that the registrants test each active ingredient per data requirements found in 40 CFR, Part 158 of the Code of Federal Regulations (CFR).^[2] Label requirements are also located in the CFR at 40 CFR, Part 156. The label is the culmination of the pesticide active ingredient testing process that may take as long as 8–10 years and costs as much as US\$35–50 million.^[3]

Fig. 1 is an example of a typical pesticide label (numbers in parenthesis below and in Fig. 1 refer to those items listed in 40 CFR, Part 156 that are required to be on all labels). Every pesticide product offered for sale, distribution, or use to the public must bear a label containing the following:

- (1) The type of pesticide, (i.e., insecticide, fungicide, herbicide, etc.).
- (2) The name, brand, or trademark under which the product is sold.
- (3) The name and address of the producer or manufacturer/registrant.
- (4) The net contents.
- (5) The product registration number (EPA Registration Number). This is a unique number found on all pesticide product labels that distinguishes a pesticide from a non-pesticide product. The EPA Registration Number indicates which company holds the registration for the product and in which sequence the product was registered.
- (6) The producing establishment registration number (EPA Establishment Number).
- (7) An ingredient statement. Each active ingredient and their respective amount (percent) must be listed on the label or supplemental label; inert ingredients are not listed separately, but a total percent is listed for all inert ingredients included in the formulation.

- (8) Signal word and symbol (if required) that convey the pesticide product's relative toxicity. A product label may display a DANGER/POISON, WARNING, or CAUTION signal word; a skull-and-crossbones symbol is required in association with a DANGER signal word if the ingredients are highly toxic orally, dermally, or through inhalation.

- (9) Warning or precautionary statements such as those related to worker protection standards, wearing of protective clothing during mixing and application, preharvest intervals (PHIs), restricted reentry intervals (REIs), hazards to wildlife, and environmental statements.
- (10) "First Aid" statement (formerly "Statement of Practical Treatment") for poisonings and spills, which usually includes a phone

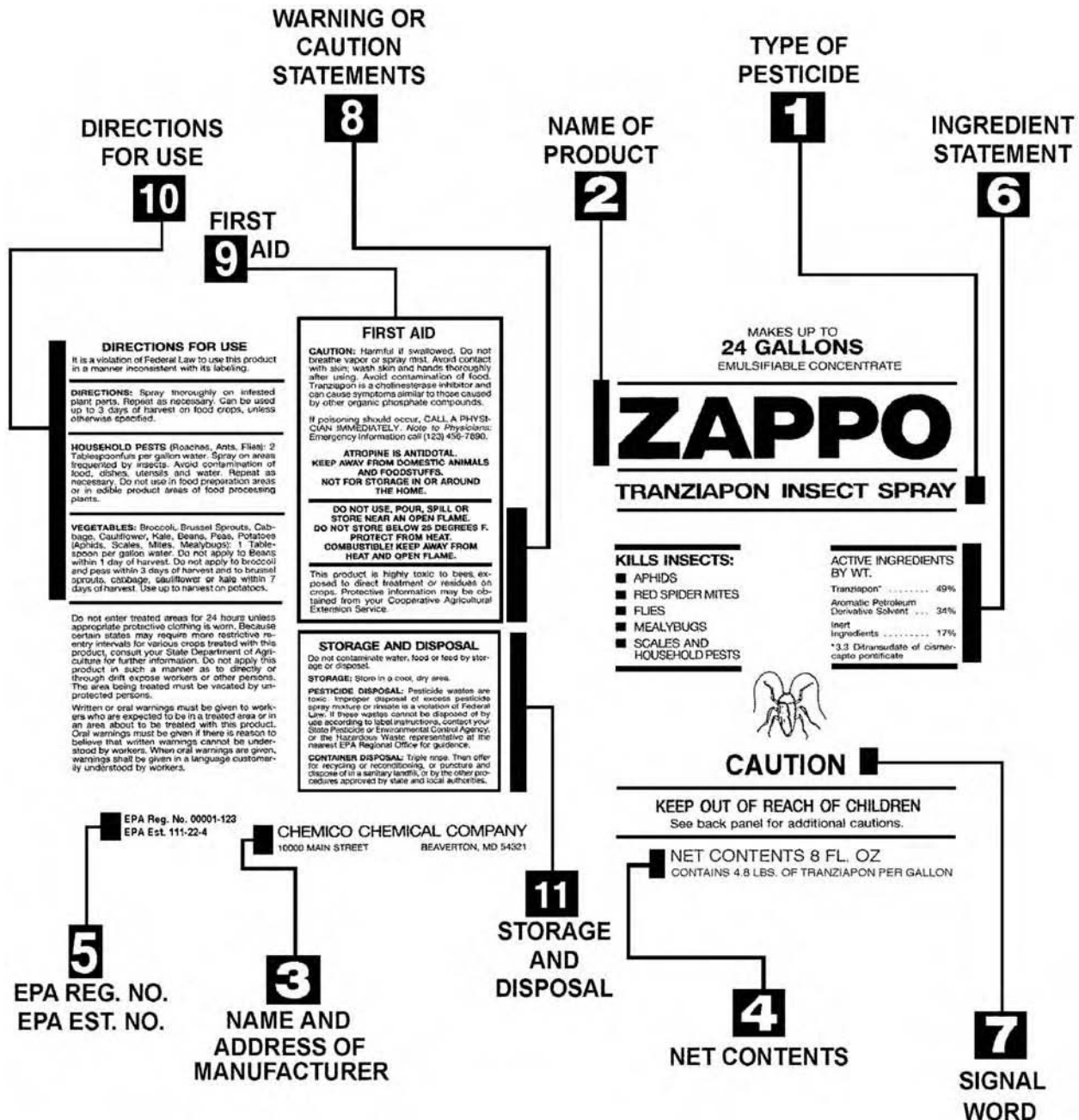


Fig. 1 Specimen label. Pesticide Management Education Program. (From *Pesticide Applicator Training Manual: CORE Manual, Northeastern Regional Pesticide Coordinators*, 2nd Ed., Cornell University, Ithaca, NY, 2000.)

number for contacting appropriate officials in case of emergencies.

- (11) Directions for use including application information (rates, compatibility, etc.), sites, and pests.
- (12) Storage and disposal statement.

In addition, pesticide products are classified as either “restricted use” or “general use.” All pesticide products that are restricted by EPA must have a “restricted-use” statement on their label. Applicators using “restricted-use” pesticides must be certified or work under the supervision of a certified applicator, as these products are usually more toxic to humans and/or the environment than those that are classified as “general use.”

TYPES OF LABELS/LABELING

Pesticide labels are associated with the type of product registration being issued by EPA and/or the individual states. The three most common types of pesticide labels that the public will encounter are the Primary Label found on the basic product registered by EPA, the Supplemental Distributor Label, and the Special Local Need (SLN) label.

Primary Label

This label appears on the container of the registrant’s basic product that has received an approved EPA registration. The primary label will contain most, if not all, of the registered uses that EPA has approved for a specific product formulation with a specific, two-part EPA Registration Number (i.e., 123-456). It should be noted here that when EPA registers a basic product, it usually registers a technical product before or at the same time. The technical product is used in the manufacture of the basic product and the end-use products intended for the public.

Supplemental Distributor Label

A second company can distribute another registrant’s basic product upon mutual agreement of both parties. A three-part EPA Registration Number (i.e., 123-456-789) on the container label will identify the product as such; the last set of numbers identifies the company distributing the product. It is the basic registrant’s responsibility to make sure that the supplemental label is in compliance with the basic product’s label. Distributor labels usually target a specific market and seldom contain all the registered uses found on the basic product label.

SLN—24(c)

Section 24(c) of FIFRA allows individual states to register additional uses of federally registered pesticides to meet an “SLN.” The SLN label provides the applicator with the proper instructions relative to the special pest problem and contains all restrictions, precautions, and limitations found on the basic product label. Food tolerance or exemption from the requirement of a food tolerance is required for all pesticide active ingredients contained in the SLN product if the use is intended to be on a food or feed commodity.

LABEL INITIATIVES

The United States and other countries have been working for over a decade to develop a Globally Harmonized System of Classification and Labeling of Chemicals (GHS). The GHS is designed to provide a common and coherent approach to defining and classifying hazards, and communicating information on labels and safety data sheets. Benefits of harmonization include enhanced protection of human health and the environment, more consistency in the classification and labeling of all chemicals, and enhancement of safer transportation, handling, and use of chemicals in transport and the workplace. The GHS is now complete and was adopted by a United Nations Committee in December 2002.^[4]

The EPA initiated a Consumer Labeling Initiative (CLI) in conjunction with other stakeholders in March 1996. The purpose of the CLI is to make pesticide labels easier to read and understand, especially for those using indoor insecticides, outdoor pesticides, and household hard surface cleaners.^[5] Several recommendations have been made via this initiative and some changes have already been incorporated on recent labels.

CONCLUSION

Pesticides are one of several tools that private and commercial applicators, food producers, and health officials have available to them for managing pest problems. In the absence of nonchemical control practices, pesticides may be the only option for a particular pest problem. Pesticide labels and related labeling provide the user with the necessary legal information for proper pest control, mixing, application, and use instructions. They also provide precautionary statements related to the environment, as well as other restrictions and limitations. In addition, individual states can require that further restrictions be placed on the EPA-registered label prior to registration in that state, especially where there are groundwater and surface water concerns. The information on the label comes from extensive

research and testing by the manufacturer/registrant and a thorough review by EPA. Notwithstanding, it is a violation of federal and state statutes when applicators use a pesticide product inconsistent with the container label and related labeling.

REFERENCES

1. U.S. Environmental Protection Agency. *Label Review Manual*; Office of Pesticide Programs; U.S. Environmental Protection Agency: Washington, DC, 1995; 1–97.
2. Office of the Federal Register, National Archives, and Records Administration. *Code of Federal Regulations*, 40 CFR Parts 150-189; U.S. Government Printing Office: Washington, DC, 2002; 1–723.
3. *What's Behind a Pesticide Label? The Pesticide Label Is Your Safety Check*; Responsible Industry for a Sound Environment (RISE): Washington, DC. http://www.pestfacts.org/behind_label.html (accessed July 2003).
4. Pont, R. *Global Harmonization of Hazard Labeling and Impacts*, Proceedings of the North American Pesticide Applicator Certification and Safety Workshop, Honolulu, HI, Aug 11–14, 2003; U.S. Environmental Protection Agency: Washington, DC, 2003.
5. U.S. Environmental Protection Agency. *Consumer Labeling Initiative, Federal Register 61 FR*; U.S. Government Printing Office: Washington, DC, 1996; 12,012–12,013.

Pesticide Reduction: Strategies

Douglas L. Murray

Peter L. Taylor

Department of Sociology, Colorado State University, Fort Collins, Colorado, U.S.A.

INTRODUCTION

Safe use generally refers to pesticide hazard reduction based on training and education in pesticide use, storage, transport, etc., with the emphasis on personal responsibility for hazard reduction through such activities as personal hygiene and reliance on personal protective equipment. These strategies for reducing the pesticide hazards, as currently implemented in the developing world, are increasingly viewed by non-governmental organizations (NGOs) and industry critics as a pesticide industry effort to placate critics while not disturbing the continued promotion of pesticides as the nearly exclusive means of pest control. However, many believe there is a potential positive role for safe use strategies, albeit only under significantly different conditions. The following discussion will describe and critique the industry's current approach to safe use and then will present an alternative approach. Many NGOs and health advocates involved in pesticide hazard reduction in the developing world favor this alternative, whose analysis has been developed in previous publications.^[1,2]

THE GLOBAL SAFE USE CAMPAIGN

Over the past decade, the Global Crop Protection Federation (GCPF), the primary association of the international pesticide industry, has taken the lead in promoting safe use through a voluntary initiative called the Global Safe Use Pilot Projects. With roughly \$1 million in funding, GCPF launched projects in Guatemala, Kenya, and Thailand in 1991. The projects focus on training and education efforts across a wide range of participants, including farmers and farmworkers, extensionists, distributors, homemakers, schoolchildren, and others. In Guatemala (the pilot country we have most closely monitored), the project has recently moved into a "self-sustaining" phase in which the Government of Guatemala turns over to AGREQUIMA, the national pesticide industry association, a 0.05% tax levied on pesticide imports to finance the industry's safe use campaign.^[3]

The pesticide industry reportedly has trained 226,000 farmers and homemakers, 2800 schoolteachers and 67,000 schoolchildren, 700 pesticide distributors, 330 technical and sales people, and 2000 physicians

and health personnel in Guatemala.^[4] The industry claims that this training has fueled a dramatic decline in pesticide poisonings over the past decade in Guatemala, an evidence of a "silent revolution" in improved pesticide use that is sweeping the developing world.

A CRITIQUE OF INDUSTRY CLAIMS

We have argued that the claims of success with industry's safe use campaign are premature and not borne out by supporting data. The industry's claim that pesticide poisonings have dropped dramatically is based largely on the documentation of illness by the Guatemalan Ministry of Health. Yet during the 1990s, the national reporting system fell into disarray as civil war and drastic cuts in government spending reduced the health sector's capacity to conduct pesticide illness surveillance. For example, in Escuintla, the department which traditionally reported the highest rates of pesticide poisoning, the pesticide reporting system was completely abandoned in the mid-1990s. National pesticide illness reports dropped steadily from roughly 2200 in 1972 to 238 cases in 1997.^[5] More reliable estimates put the pesticide poisoning rate in the latter half of the 1990s at nearly 10,000 cases annually.^[5]

Industry claims of the success of safe use are subject to question on other grounds as well. Merely reporting the number of people trained does not represent any indication of an impact on pesticide hazards. It confounds outputs (numbers trained) with outcomes (decreased pesticide poisoning). Furthermore, safe use training as employed in Guatemala assumes that a transfer of knowledge leads in a linear fashion to changes in behavior. However, behavioral change, even where it is actually demonstrated, can often be a temporary artifact of the training process rather than a lasting result. Once the participants are no longer observed by their trainers, they frequently revert to traditional and hazardous practices.

Possibly even more critical to the inadequacy of the safe use campaign is the lack of recognition of why hazardous practices occur in the developing world. The campaign focuses almost entirely on telling people why they should behave differently and more rationally. However, in many instances, hazardous practices

reflect rational choices made by pesticide users seeking to maximize profits, save time, or even to avoid losing jobs. An example of this problem occurred in Honduras in the late 1980s. A group of farmworkers was poisoned after applying carbofuran to a melon patch with their bare hands, then eating lunch without washing. They were not provided safety equipment such as rubber gloves, and no water was provided for washing. Washing their hands would have required leaving the field, thus losing their brief break for lunch. Leaving the field or requesting safety equipment from the farm manager could also have jeopardized their tenuous employment as day laborers.^[6] Unsafe behavior is frequently a reflection of the structural and social conditions of work in the developing world, and not simply a lack of knowledge or care on the part of workers.

Finally, the industry approaches the pesticide problem in their literature in contradictory ways that undermine the effective pursuit of hazard reduction. Often, the literature portrays the problem as one exaggerated by public perception rather than accepting that pesticide poisonings are a serious and persistent problem.^[7] Such arguments undermine the efforts of NGOs and others to raise public awareness and change government policies in ways that might bring the safe use of pesticides under better control.

MAKING SAFE USE WORK: AN ALTERNATIVE APPROACH

A more effective approach to pesticide hazard reduction, and one that would make safe use campaigns much more reliable, is based on hazard reduction strategies employed in industrial sectors in Europe and the United States. This approach relies on a hierarchically organized series of interventions. When applied to the pesticide problem in the developing world, this strategy is pursued as follows.

Hazard Elimination

A number of the most toxic and problematic pesticides in use in the developing world must be removed from use prior to subsequent and subordinate problem-solving measures. In regions such as Central America, this would primarily involve the elimination of a number of pesticides classified by the World Health Organization as Category 1 chemicals, deemed highly or extremely hazardous.^[8,9] These pesticides are dangerous under ideal conditions, and, in the developing world, their use results in hundreds of thousands of poisonings annually. The removal of these products should occur through a combination of voluntary

withdrawals of registration by the pesticide industry and government regulatory actions.

Substitution of Safer Alternatives

Once eliminated, there will be a need for alternative pest control measures. This will require the use of less toxic pesticides in some cases, and in others, the promotion of Integrated Pest Management (IPM), organic farming, and other alternative approaches to pest control. The pesticide industry thus far has failed to enter into the substitution process except to promote their individual product lines. In the case of IPM, the pesticide industry has defined this alternative approach largely in pesticide-dependent ways, focusing on economic threshold analysis that simply puts off pesticide application until the threat of economic loss reaches a certain level, at which point the farmer is encouraged to return to the near exclusive reliance on pesticides. Alternative approaches to IPM that depend on non-chemical and biological control measures are not seriously entertained by the pesticide industry.

In many cases, governments in the developing world have been passive at best in promoting IPM and other alternatives. Often, the Ministries of Agriculture in these countries, where primary pesticide regulatory responsibility is located, have deferred to the pesticide industry's views on promoting alternatives. A more proactive role for government health and environmental agencies, combined with more pressure from civil society, will likely be necessary to achieve effective implementation of pesticide problem solving at this level.

Administrative Controls

Once the most hazardous pesticides have been eliminated and safer alternatives are adopted, administrative measures such as training make more sense. Safe use can be much more effective once the greatest hazards are gone. However, determining the efficacy of training should not be left up to the pesticide industry. It will likely fall back into the hands of the industry's public relations people instead of being developed and evaluated by external parties such as NGOs.^a Using focus groups to develop training and related interventions based on the participatory identification of problems, including structural problems exemplified

^aFor a well-done exception to most industry evaluations of safe use, see Ref.^[10]. However, this study is also overly focused on the individual behavioral and psychological analysis common to industry evaluations and largely ignores the structural and social factors influencing individual behavior.

by the Honduran case above, will give this level of intervention far more impact on pesticide hazards.

Personal Protective Equipment

Finally, the use of safety equipment can be considered. But again, this makes sense only after the most hazardous chemicals have been eliminated, less toxic alternatives implemented, and training and other administrative measures adopted. Safety equipment in the industrial setting has long been recognized as the last and least effective means of assuring safety, and the pesticide industry and governments should adopt a similar perspective for the agricultural sector in the developing world.

CONCLUSION

Safe use, as currently pursued, is not a viable means for significantly reducing pesticide hazards. It is time for the pesticide industry and governments in the developing world to step forward and take more serious measures to reduce pesticide hazards. Non-governmental organizations will continue to pressure governments and the pesticide industry to go beyond palliative efforts such as the current safe use campaigns and pursue more serious responses along the lines of the alternative strategy described above.

ACKNOWLEDGMENT

The authors have worked with a variety of NGOs in the developing world over the past two decades, including CARE International, OXFAM, Greenpeace, the Pesticide Action Network, and others, on pesticide

hazard reduction, safe use training, promotion of alternative pest control practices, and related strategies of improving safety and productivity in agriculture. However, the analysis in this article does not represent the policy of any particular NGO.

REFERENCES

1. Murray, D.L.; Taylor, P.L. Claim no easy victories: evaluating the pesticide industry's global safe use campaign. *World Dev.* **2000**, *28* (10), 1735–1749.
2. Murray, D.L.; Taylor, P.L. *Beyond Safe Use: Challenging the International Pesticide Industry's Hazard Reduction Strategy*; Gatekeeper Series no. 103; International Institute for Environment and Development, 2001.
3. Hurst, P. *The Global Pesticide Industry's 'Safe Use and Handling' Training Project in Guatemala*; International Labour Organization: Geneva, 1999.
4. GIFAP. *Proteccion de Cultivos: Proyectos de Uso y Manejo Seguro en America Latina*; Grupo Internacional de Asociaciones Nacionales de Fabricantes de Productos Agroquimicos: Guatemala City, n.d.
5. de Campos, M.; Finkelman, J. *Situacion Actual del Uso y Manejo de Plaguicidas en Guatemala*; Organizacion Panamericana para la Salud: Guatemala, 1998.
6. Murray, D.L. *Cultivating Crisis: The Human Cost of Pesticides in Latin America*; University of Texas Press: Austin, 1994.
7. Renan, A.A.; Felipe, A.C. El sentido comun y los plaguicidas. *Tierra Fertil.* **1998**, *2*.
8. International Programme on Chemical Safety (IPCS). *Classification of Pesticides by Hazard and Guidelines to Classification, 2000–2002*; World Health Organization: Geneva, 2001.
9. www.pesticideinfo.org.
10. Atkin, J.; Leisinger, K.M. *Safe and Effective Use of Crop Protection Products in Developing Countries*; CABI Publishing: New York, 2000.

Pheromone Traps and Trapping

Jenn-Sheng Hwang

Department of Applied Toxicology, Taiwan Agricultural Chemicals and Toxic Substances Research Institute, Council of Agriculture, Executive Yuan, Taiwan, Republic of China-Taiwan

Chau-Chin Hung

Taiwan Agricultural Chemicals and Toxic Substances Research Institute, Council of Agriculture, Executive Yuan, Republic of China

INTRODUCTION

Since the first pheromone was identified in 1959 by Butenandt et al.,^[1] so far, more than 1000 insect pheromones have been identified and synthesized for many of the major agricultural and forest pest species from the orders Lepidoptera, Coleoptera, Homoptera, Diptera, Hymenoptera, Isoptera, Blattodea, and Hemiptera. Among them, about 300 sex pheromones (also called sex attractants) and related compounds are used worldwide in integrated pest management (IPM) programs.

The main practical applications of insect pheromones in pest management include: 1) survey and monitoring with pheromones as attractants for early-warning, quarantine work, timing of control measures, population trends, dispersion, risk assessment, and assessment of the effects of control measures; 2) mass trapping; 3) mating disruption; and 4) crop trapping, etc. Except mating disruption and crop trapping, trap designs are needed to be used either for the detection or monitoring of the pests or for mass trapping in control programs. Several components of developing a pheromone trapping system and the trapping efficiency affected by some factors will be discussed with regard to different application objectives. As for the other pheromone traps used in IPM programs, they have been discussed in other sections of this book.

PHEROMONE LURE

The basic components of a pheromone trapping system include pheromone lure (attractant), trap design, and trap employment. The chemical composition of the attractant will depend on the species to be trapped and is usually an accuracy of blend ratio of mixture of synthetic compounds identical to the identified natural pheromones. Lists of identified pheromones and attractants are available in the literature^[2] and from supply organizations. The required purity of individual components and the dose and formulation of the attractant used are critical to determine the bioactive (attractancy), lure persistence (longevity), and release

rate and active distance (attractive range), as well as use objective.

In general, several micrograms or milligrams of dose of pheromones are dispensed in cotton wicks, rubber septa, polyethylene and polyvinyl chloride vials, capillary or hollow fibers, and trilaminates, etc. For example, 1 mg of (*Z*)-3-dodecen-1-ol (*E*)-2-butenate formulated into PVC microtube was attractive to sweet potato weevil, *Cylas formicarius*, for more than 2 months and is cheaper than the rubber septum;^[3] and the rubber septum containing 0.5–1 mg of *Z*-8-dodecenyl acetate caught carambola fruit borer, *Eucosma notanthes*, consistently for at least 6 months in orchards.^[4]

TRAP DESIGNS

Trap design is essential in developing trapping systems, and the variety of pheromone traps used in IPM programs depends on diverse insect behavior and different trapping purposes. Traps used in field trapping program need to be practical as well as efficient; they should be cheap and easy to maintain and deploy, and resistant to weather as sun, rain, and strong wind.

Sticky trap is the most common type of traps in use, and it employs a sticky surface to retain or immobilize the attracted insects. The most common sticky traps are the vertical sticky plate, the delta trap, the tent trap, the cylinder trap, the paired board trap, and the wing trap (Fig. 1A–F), etc. Sticky traps are generally more efficient at catching attracted insects than are other types of traps, and traps with exposed sticky surfaces are more efficient than traps with sticky surfaces enclosed,^[5] because more insects come into contact with the sticky surface.

When the sticky surface is aged and becomes saturated with captured insects and debris, its ability to retain new arrivals is reduced or even eliminated. Generally, the sticky trap used in the field needs to be renewed in 1 or 2 weeks depending on the population level of the target species and on weather conditions. Although the sticky trap used in IPM programs is usually costly and laborious, it is suitable for catching

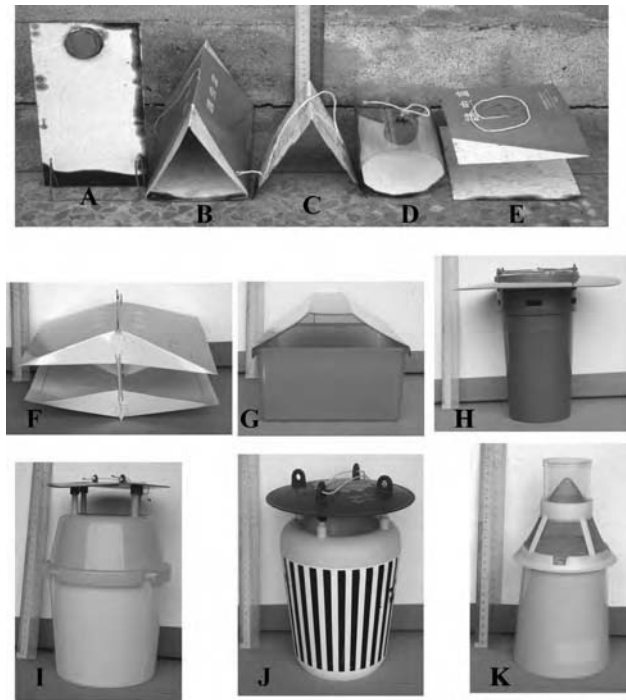


Fig. 1 Various types of pheromone trap designs. (A) vertical sticky plate; (B) delta trap; (C) tent trap; (D) cylinder trap; (E) paired board trap; (F) wing trap; (G) water trap; (H) gypsy moth trap; (I) funnel trap; (J) black stripe moth trap; (K) infield trap or Dickerson trap or Hardee trap for boll weevil.

small insect pests such as male mealy bug and scale insects, white fly, leaf miner, small moths such as tortricids, gelechiids, and plutellids, as well as small stored product insects.

The water trap is also a common type of trap and is composed of two parts, an upper cover and a lower container of basin or tray (Fig. 1G). The pheromone lure is attached to the underside of the cover, and the dilute detergent water is in the container for drowning the attracted insects. The container of water trap is usually large enough to retain the insects and is also effective, especially when the insects are searching for pheromone plume, and also when seeking drinking water. The water traps need to be periodically refilled.

A variety of non-sticky traps with large capacity and unvaried efficiency offer alternative approaches. Many different types of no-exit traps with entrance ports have been developed for larger moths, beetles, and dipterous insects. These no-exit traps are provided with optimal openings for admission of the attracted insects. The optimal orifice diameter for trapping most lepidopterous species is about twice the average thorax width of the males.^[5]

To make available pheromone traps for use in IPM programs, many companies in cooperation with governmental researchers have developed various trap

designs for different insect species and then manufacture these products for commercial use. A field-researched, -tested, and -patented trap for gypsy moth (Fig. 1H) has been commercialized by SureFire in the United States and Canada, and is like the primitive “milk carton” trap. A funnel trap (Fig. 1I) made by Biosense is particularly suitable for larger lepidopterous pests such Noctuids which often occur in large numbers. And the AgriSense black-stripe moth trap (Fig. 1J) is also recommended for use as a permanent system for the monitoring of moths of stored products. The “infield trap” or Dickerson trap Hardee trap (Fig. 1K), similar to the original “Leggett trap” design, is marketed by Scentry and is widely used in boll weevil monitoring and mass trapping projects in the United States and abroad. For forest *Scolytus* and *Dendroctonus* bark beetles, the Lindgren multiple funnel trap from PheroTech (Fig. 2A) is recommended. The Madalacol trap (Fig. 2B) is used to catch pine sawyer, *Monochamus alternatus*, in Japan.

For fruit-fly species such as the Mediterranean, Oriental, and Queensland fruit fly and melon fly, the dome trap (Fig. 2C), a modified version of the original McPhail trap, is ideally suited for baiting with food lures such as protein hydrolysate, molasses, or ammonium salt solutions, and then deployed to catch females and males. It is made as a two-piece plastic trap with a clear dome-shape cover and is easier to clean. The yellow base provides a visual attractant to complement the food lure inside. The lantern trap

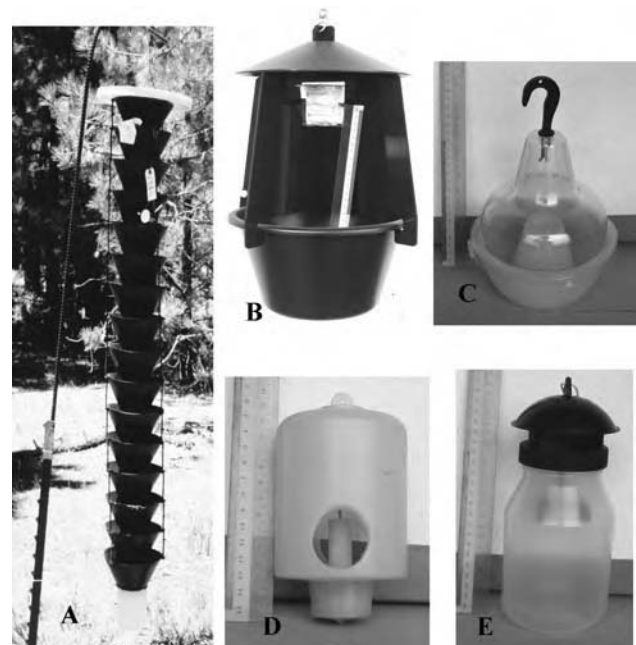


Fig. 2 Various types of pheromone trap designs. (A) Lindgren multiple funnel trap for bark beetles; (B) Madalacol trap for pine sawyer; (C) dome trap; (D) lantern trap; (E) fruit fly trap.

(Fig. 2D) baited with poisoned methyl eugenol has been used in fruit-fly monitoring programs, and, currently, the fruit fly trap (Fig. 2E) with methyl eugenol inside is the preferred one.

In Taiwan, different types of no-exit traps (also called dry traps) have been designed and made out of used plastic soda bottles which are low cost, easy to use, and efficient. For trapping tobacco cutworm, *Spodoptera litura*, and/or beet armyworm, *S. exigua*, a no-exit polyethylene terephthalate (PET) soda bottle trap (Fig. 3A) had been developed with a length of 3 cm and a width of 0.3–0.5 cm of punctured entrance ports, with an additional bottle at the bottom of the trap that acts as a funnel trap to prevent the moths from escaping.^[6] Its catching capacity exceeds several thousand moths, in contrast to the sticky wing trap which becomes saturated with about 30–40 moths. A double-funnel PET bottle trap (Fig. 3B) was designed for trapping sweet potato weevil.^[3] The three-layered funnel-type PET bottle trap (Fig. 3C)



Fig. 3 Various types of pheromone trap designs. (A) PET soda bottle trap; (B) double-funnel PET bottle trap; (C) three layers funnel-type PET bottle trap; (D–F) commercialized dry plastic traps for many species of *Lepidoptera* and sweet potato weevils.

has been developed with 16 openings of 0.6–0.8 cm and with a pheromone lure placed 5 cm above the opening. This trap is effective at trapping carambola fruit borer.^[4] These no-exit trap designs are practical, effective, and cheap at a cost of only 10 cents per trap when made by hand out of used plastic soda bottles, and have been widely used by farmers in Taiwan, as well as the commercially produced plastic traps (Fig. 3D–F), which cost only U.S.\$2–3.

Sometimes the no-exit trap is not as efficient as a sticky trap and uses a vaporous insecticide (dichlorvos) to kill the trapped insects; however, to date, little attention has been given to the possibility that the insecticide odor repels the insect before it can enter the trap. The funnel-type PET bottle trap baited with the sex pheromone of the carambola fruit borer and with a dichlorvos strip inside reduced trap catch by 13.4%.^[4] But the funnel trap with a dichlorvos strip placed inside did not affect sweet potato weevil capture but could prevent captured weevils from escaping.^[7]

The size of the trap may be important, especially with traps relying on a sticky retentive surface, but the operation convenience of the trap also needs to be considered in determining the trap size. The traps with funnel diameters of 5.5 to 25.5 cm showed no difference in the number of male sweet potato weevils captured.^[8]

Trap color has been regarded as important in affecting the catch. In the day-flying gypsy moth it has not been shown to influence catch,^[9] but in the morning-flying carambola fruit borer, clear (transparent) PET bottle traps were more effective than green traps.^[4] It is reported that the color of the cylinder-type sticky trap does not influence trap catch in the citrus mealybug, *Planococcus citri*,^[10] but the clear (transparent) funnel traps are more effective at capturing sweet potato weevils than green-colored traps,^[7] and the yellow cylinder-type sticky trap is more effective at trapping striped flea beetle, *Phyllotreta striolata*, than other colors.^[11]

TRAP DEPLOYMENT

There are three main considerations in trap placement: trap height and position with respect to vegetation, and trap density. Traps for bark beetles placed in open areas were consistently more effective than those near trees. Male codling moth trap catch is highest when traps are positioned near the top of the apple tree canopy.^[9] For trapping litchi fruit borer, *Conopomorpha sinensis*, the wing sticky trap hung on the canopy height inside the litchi trees caught more male moths than those placed at lower position and outside the litchi trees.^[12] The dry PET bottle trap for catching tobacco cutworm was recommended for installation

at a height of 0.5 m up to 1.5 m above ground in legume fields.^[6] The double-funnel traps with a pheromone lure placed 4 cm above the funnel caught significantly more sweet potato weevils than those traps with pheromone lure placed 0 or 8 cm above the funnels.^[7] And no significant difference was found in trap catch when traps with the top of the funnel placed 4, 8, or 16 cm above the sweet potato canopy were used.^[7] However, in practice the trap height should be adjustable to the height of the vegetation.^[9,13]

Trap density (trap spacing) can be determined by the active distance of a pheromone-baited trap, insect-searching ability, and the trapping objective, but is a much more complex problem. In area-wide monitoring projects, one trap per 1–10-ha field may be enough to detect the occurrence of insects. For mass-trapping programs, existing experimental data indicate that high trap densities are often needed to obtain high levels of population suppression. The active distance of the sex pheromone trap of the sweet potato weevil was determined to be about 10–15 m,^[3] and 40 funnel traps were suggested for use in a 1-ha field for mass trapping.^[14] Ten traps per hectare of legume fields were recommended for both mass trapping and pest monitoring of the tobacco cutworm.^[6]

Wind direction may affect the trap catch.^[9,13] Generally, the trap placed in the upwind location and crossing the wind direction may be preferable to catch more insects. Wind speed also influences the flight behavior of the insect and therefore the optimum trap height. At high wind speed, searching males may fly close to the leeward side of the host plants and may even cease flight at strong wind.

THE MERIT AND SHORTCOMING OF PHEROMONE TRAP

The merit of pheromone traps are that they catch selective insect species at low population densities, no power required as compared to light trap, and their relative ease of use, which makes them ideal for use by farmers, foresters, and growers who are not trained entomologists.

The major shortcomings of pheromone-monitoring traps are the difficulty of relating trap catches to pest density or crop damage levels, because the sampling efficiency of some traps seems to change with pest density and weather conditions. Several authors have stated that trap efficiency declines with increasing population density, and this has been attributed to competition with wild females.^[5,9,13] The weather, especially temperature, humidity, and wind, undoubtedly affects pheromone trap catches as they depend on the behavioral responses of the insects. This may be a disadvantage or an advantage depending on the

objective, because weather affects male response to the pheromone and may also affect female flight and egg-laying activities in the field.

CONCLUSION

Overall review of available pheromone lures and trap designs indicates that there is considerable variation in their effectiveness at trapping different insect species. However, application technology has been developed and has contributed to many phases of insect management, including surveillance, suppression, and program evaluation, as well as to basic research on the biology, behavior, and population dynamics of insect pests.

Development of more effective, inexpensive, and practical pheromone trapping systems should continue to be studied, and direct observation of male behavior in relation to trap design is needed to determine true trap efficiency. If usual relationships between trap catch and population density or crop damage can be established, we can then look for a trapping system that will be more practical for widespread field use. However, the use of traps containing sex pheromone and related chemicals has become an important component in IPM programs.

ACKNOWLEDGMENTS

The assistance of Miss Chia-ying Liu in the preparation of the manuscript and the figures is gratefully acknowledged.

REFERENCES

1. Butenandt, A.T.; Beckman, R.; Stamm, D.; Hecker, E. Über den sexuallockstoff des seidenspinners *Bombyx mori*, Reidarstellung und konstitution. *Z. Naturforsch.*, **B 1959**, *14*, 283–284.
2. Inscoc, M.N.; Leonhardt, B.A.; Ridgway, R.L. Commercial availability of insect pheromones and other attractants. In *Behavior-Modifying Chemicals for Insect Management*; Ridgway, R.L., Silverstein, R.M., Inscoc, M.N., Eds.; Marcel Dekker Inc.: New York, 1990; 631–715.
3. Hwang, J.S.; Hung, C.C.; Yen, Y.P. Evaluation on formulations of synthetic sex pheromone and trap designs for trapping sweet potato weevil, *Cylas formicarius elegantulus* (Summers). *Chin. J. Entomol.* (Taiwan) **1989**, *9*, 37–43.
4. Hwang, J.S.; Hung, C.C. Formulations of sex attractant and trap designs for trapping carambola fruit borer, *Eucosma notanthes* Meyrick. *Plant Prot. Bull.* (Taiwan) **1994**, *36*, 31–40.

5. Lanier, G.M. Principle of attraction–annihilation: mass trapping and other means. In *Behavior-Modifying Chemicals for Insect Management*; Ridgway, R.L., Silverstein, R.M., Inscoc, M.N., Eds.; Marcel Dekker Inc.: New York, 1990; 25–45.
6. Lee, S.C. Capture efficiency of various sex pheromone traps for *Spodoptera litura*. *Chin. J. Entomol. (Taiwan)* **1987**, *7*, 87–94.
7. Hwang, J.S. Integrated control of sweet potato weevil, *Cylas formicarius*, with sex pheromone and insecticide. *Ext. Bull. - ASPAC, Food Fertil. Technol. Cent.* **2001**, *494*, 1–13.
8. Proshold, F.I.; Gonzalez, J.L.; Asencio, C.; Heath, R.E. A trap for monitoring the sweet potato weevil using pheromone or live females as bait. *J. Econ. Entomol.* **1986**, *79*, 641–647.
9. Carde, R.T.; Elkinton, J.S. Field trapping with attractants: methods and interpretation. In *Techniques in Pheromone Research*; Hummel, H.E., Miller, T.A., Eds.; Springer-Verlag: New York, 1984; 111–129.
10. Hwang, J.S.; Chu, Y.I. The development of sex pheromone traps for the citrus mealybug, *Planococcus citri* (Risso). *Plant Prot. Bull. (Taiwan)* **1987**, *29*, 297–305.
11. Hung, C.C.; Hwang, J.S. Influence of cylinder-type sticky traps baited with different mustard oil lures on *Phyllotreta striolata*. *Chin. J. Entomol. (Taiwan)* **2000**, *20*, 201–214.
12. Hwang, J.S.; Hung, C.C.; Yen, Y.P.; Chen, C.N. Sex attractant and trap designs for trapping litchi fruit borer, *Conopomorpha sinensis* Bradley. *Plant Prot. Bull. (Taiwan)* **1996**, *38*, 129–136.
13. Wall, C. Monitoring and spray timing. In *Insect Pheromone in Plant Protection*; Jutsum, A.R., Gordon, R.F.S., Eds.; John Wiley and Sons: New York, 1985; 39–66.
14. Hwang, J.S.; Hung, C.C. Evaluation of the effect of integrated control of sweet potato weevil, *Cylas formicarius*, with sex pheromone and insecticide. *Chin. J. Entomol. (Taiwan)* **1991**, *11*, 140–146.

Pheromone Use in Integrated Pest Management of Stored Products

Pasquale Trematerra

Department SAVA, University of Molise, Campobasso, Italy

INTRODUCTION

The use of pheromones is one of the most promising techniques aimed at the control of stored-product pests Coleoptera and Lepidoptera. The use of these substances can lead to a drastic reduction of chemical treatments, thus determining remarkable economic advantages and improvement of product quality, protecting goods from residual insecticides noxious to the consumer. In recent years, considerable progress has been made in monitoring and control of stored-product insects by pheromones also used in mass trapping, attracticide (lure and kill), and mating disruption methods.

The development of integrated pest management (IPM) programs has been considered by the food industry for both raw and processed commodities.^[1] The IPM concept emphasizes the integration of disciplines and control measures including biological enemies, cultural management, sanitation, proper temperature utilization, and pesticides into a total management system aimed at the prevention of pests from reaching damaging levels. The food industry will need to use IPM programs more extensively in the future to satisfy the increased demands of consumers and regulatory agencies for reduced use of pesticides.

In that context, considerable progress has been made in the use of pheromones for monitoring and control (by mass trapping, attracticide, and mating disruption) of stored-product pests (Table 1).^[2-6]

MONITORING

Pheromone traps in stored insect management can be used to detect both the presence and the density of pests. They are useful in defining areas of pest infestation, particularly where the overall distribution and life cycle are poorly understood. Their purpose is to achieve a more accurate control and to limit insecticide use.

Pheromone traps are generally effective when pest numbers are very low and they can be used qualitatively to provide an early warning of pest incidence (Figs. 1 and 2). To successfully capture attracted pest insects, a trap has to be escape proof; this can be

achieved by a sticky surface to which the trapped insects become irreversibly attached or by some kind of funnel or pitfall systems.^[7] Designs of traps for beetles (*Cryptolestes* spp., *Lasioderma*, *Oryzaephilus* spp., *Prostephanus*, *Rhyzopertha*, *Sitophilus* spp., *Stegobium*, *Tribolium* spp., *Trogoderma*, etc.) and moths (*Ephestia* spp., *Plodia*, *Sitotroga*, etc.) infesting stored products have been developed, generally on an empirical basis.

A list of the factors, known to affect trap catch, which should be addressed during the design, execution, and reporting of trapping studies was reported.^[8] Typical recommendations provide for the placement of a gridwork of traps and their monitoring for the capture of insects at regular time intervals.^[9] Optimization of traps and lures will allow the realization of new computer-based methods aimed at the organization and interpretation of data, and will make it easier to face pest attacks properly.^[10]

MASS TRAPPING

In the case of female-produced sex pheromones, only males are trapped. Hence, any attempt to suppress the population by trapping males would require a sufficient number of trapped males so that nearly all females would go unmated. Theoretical considerations of mass trapping males take into account the density of males in the population and the potential number of matings a male is able to secure in its lifetime. If a male can mate with 10 females in a lifetime, as is the case for *Plodia interpunctella*, then up to 90% of the male population can be trapped without affecting the number of mated females as well as the subsequent larval generation. Under high population levels, the rate of female encounters would be high and mass trapping more difficult to achieve. However, under low population levels, males would locate females less frequently and intensive trapping could conceivably reduce male populations to biologically significant levels.

Proper experiments of mass trapping are not easy to conduct due to inadequate controls or poor replication. However, various studies have reported success in the control of *Ephestia cautella* in United States, *P. interpunctella* in a storage room for vegetable and

Table 1 Stored-product insects for which pheromones have been identified and available

| Species | References ^a | Sex producing and pheromone type | Availability of traps and lures |
|----------------------------------|------------------------------|----------------------------------|---------------------------------|
| <i>Coleoptera</i> | | | |
| <i>Acanthoscelides obtectus</i> | Mori et al. (1981) | Male, sexual | No |
| <i>Anthrenus flavipes</i> | Sharma et al. (1991) | Female, sexual | Yes |
| <i>Anthrenus verbasci</i> | Kuwahara and Nakamura (1985) | Female, sexual | Yes |
| <i>Attagenus brunneus</i> | Fukui et al. (1977) | Female, sexual | No |
| <i>Attagenus unicolor</i> | Silverstein et al. (1967) | Female, sexual | Yes |
| <i>Callosobruchus chinensis</i> | Mori et al. (1983) | Female, sexual | No |
| <i>Cryptolestes ferrugineus</i> | Boden et al. (1993) | Male, aggregation | Yes |
| <i>Cryptolestes pusillus</i> | Abdukakharov et al. (1997) | Male, aggregation | Yes |
| <i>Cryptolestes turcicus</i> | Millar et al. (1985) | Male, aggregation | Yes |
| <i>Dermestes maculatus</i> | Levinson et al. (1978) | Male, aggregation | No |
| <i>Lasioderma serricorne</i> | Mori and Watanabe (1985) | Female, sexual | Yes |
| <i>Oryzaephilus mercator</i> | Odinokov et al. (1993) | Male, aggregation | No |
| <i>Oryzaephilus surinamensis</i> | Boden et al. (1993) | Male, aggregation | No |
| <i>Prostephanus truncatus</i> | Hodges et al. (1984) | Male, aggregation | Yes |
| <i>Rhyzopertha dominica</i> | Razkin et al. (1996) | Male, aggregation | Yes |
| <i>Sitophilus granarius</i> | Mori and Ishikura (1989) | Male, aggregation | Yes |
| <i>Sitophilus oryzae</i> | Pilli (1993) | Male, aggregation | Yes |
| <i>Sitophilus zeamais</i> | Pilli (1993) | Male, aggregation | Yes |
| <i>Stegobium paniceum</i> | Matteson and Mann (1994) | Female, sexual | Yes |
| <i>Tribolium castaneum</i> | Odinokov et al. (1991a) | Male, aggregation | Yes |
| <i>Tribolium confusum</i> | Odinokov et al. (1991b) | Male, aggregation | Yes |
| <i>Trogoderma glabrum</i> | Mori et al. (1985) | Female, aggregation-sexual | Yes |
| <i>Trogoderma granarium</i> | Pawar et al. (1993) | Female, aggregation-sexual | Yes |
| <i>Trogoderma inclusum</i> | Mori et al. (1978) | Female, aggregation-sexual | Yes |
| <i>Trogoderma variabile</i> | Mori et al. (1978) | Female, aggregation-sexual | Yes |
| <i>Lepidoptera</i> | | | |
| <i>Corcyra cephalonica</i> | Naoshima et al. (1991) | Male, sexual | No |
| <i>Ephestia cautella</i> | Odinokov et al. (1991c) | Female, sexual | Yes |
| <i>Ephestia elutella</i> | Odinokov et al. (1991c) | Female, sexual | Yes |
| <i>Ephestia kuehniella</i> | Odinokov et al. (1991c) | Female, sexual | Yes |
| <i>Plodia interpunctella</i> | Odinokov et al. (1991c) | Female, sexual | Yes |
| <i>Sitotroga cerealella</i> | Odinokov et al. (1991d) | Female, sexual | Yes |
| <i>Tineola bisselliella</i> | Yamaoka et al. (1985) | Female, sexual | Yes |

^aFor chemical synthesis, identification, or analysis of pheromones.

flower seeds in France, *E. kuehniella* in some Italian mills, *Lasioderma serricorne* and *P. interpunctella* in two food warehouses in Hawaii, and *L. serricorne* in tobacco stores in Greece and in a Hawaiian bakery.^[6,11–13]

Mass trapping both sexes of a population using aggregation pheromones should be more effective than mass trapping only males. Aggregation pheromones are known from several beetle species that infest stored products, but few studies have been conducted to suppress populations of these insects.

ATTRACTICIDE

The attracticide (lure and kill) concept-based method involves using a pheromone or other attractive

semiochemical to lure insects to a specific point source or an area whereby they contact a toxicant that causes a rapid kill or contamination with some kind of pathogen. This method is in some ways analogous to mass trapping, although many more insects are affected because the attracticide is broadcast over a large area and the killing effect is not limited to individual traps.

In stored-products protection, the attracticide concept is promising in flour mills and confectionary industries in the control of *E. kuehniella* and *E. cautella*. In Italian mills, Mediterranean flour moth males were successfully lured to laminar dispensers, baited with 2 mg of (ZE)-9,12-tetradecadien-1-yl acetate (TDA) and treated with 5 mg of cypermethrin; this caused a marked decrease in moth population. This technique led to a drastic reduction in chemical treatments with subsequent economic and qualitative advantages.^[14]



Fig. 1 Inspection of a pheromone sticky trap (delta type) used in the monitoring of moths infesting stored products.

Another attracticide method utilized pheromones in an inoculation device containing a pathogen (a protozoan in the control of *Trogoderma glabrum*, a granulosis virus against *P. interpunctella*).^[2,15]

MATING DISRUPTION

The mechanisms involved in mating disruption may consist of one or a combination of any of the following: The constant exposure of the insect to a relatively high level of pheromone leads to the adaptation of the antennal receptors; a sufficiently high background level of the applied pheromone masks the natural pheromone plumes; the synthetic plume pheromone is applied in a relatively large number of discrete sources. The limitations and theoretical bases of mating disruption are similar to those for mass trapping of males.

Several successful experiments have been reported in mating disruption of *E. cautella* and *P. interpunctella* both in the laboratory and in simulated field situation, and *E. kuehniella* in a food industry. Other mating inhibitory compounds are known for Coleoptera *L. serricornis* and *Stegobium paniceum*.^[6,16]

Mating disruption is a potentially effective pheromone-based control method for storage insects, but more data are necessary in order to reduce the quantity of pheromones used and the risk of their residues in food.

FUTURE PROSPECTS

In stored-product protection, different tolerance thresholds should be established for the various pests depending on their economic impact and on the “filere

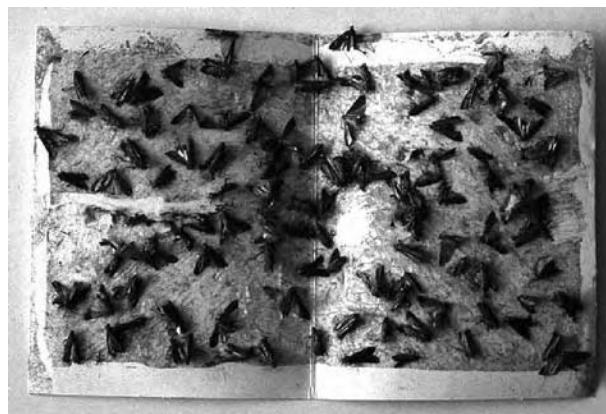


Fig. 2 Sticky surface of a pheromone trap with adults of *E. kuehniella* trapped in a flour mill.

place” where they are found. For example, a limited number of insects can be tolerated at times in a storehouse containing raw materials, but in food-processing plants and storehouses containing finished products, the threshold must be necessarily zero. The utilization of pheromones and other semiochemicals could lead to a drastic reduction of chemical treatments with consequent economic and qualitative advantages, protecting goods from residual products noxious to the consumer.

Crucial factors for IPM in stored products include understanding factors that regulate systems, monitoring insect populations, maintaining good records, and using this information to make sound management decisions. In that context, “insectistasis”^[17] can be readily achieved by continual supervision of environments by attractant traps in combination with a limited number of curative measures appropriately timed.

New tools have been developed for detecting insects in stored products, estimating insect population growth, and administering fumigants as well as natural methods of insect control such as grain temperature manipulation. Existing or potential new technologies for detecting the presence of insects and estimating insect population levels include pheromone traps, sampling devices, acoustic sampling methods, and chemical tests that detect live or dead insects through the presence of enzymes.^[18] Computer-assisted decision support systems have also been developed, which estimate insect population growth and spatial distribution of insects as a function of the environmental factors.^[19,20]

REFERENCES

1. Hagstrum, D.W.; Flinn, P.W. Integrated pest management. In *Integrated Management of Insects in Stored Products*; Subramanyam, B., Hagstrum, D.W., Eds.; Marcel Dekker: New York, 1996; 399–408.

2. Burkholder, W.E. Practical use of pheromones and other attractants for stored-product insects. In *Behavior-Modifying Chemical for Insect Management*; Ridgway, R.L., Silverstein, R.M., Inscoc, M.N., Eds.; Marcel Dekker: New York, 1990; 497–516.
3. Chambers, J. Overview on stored-product insect pheromones and food attractants. *J. Kans. Entomol. Soc.* **1990**, *63*, 490–499.
4. Phillips, T.W. Semiochemicals of stored-product insects: research and applications. *J. Stored Prod. Res.* **1997**, *33*, 17–30.
5. Trematerra, P. Integrated pest management of stored-product insects: practical utilization of pheromones. *Anz. Schaedlingskd., Pflanzenschutz, Umweltschutz* **1997**, *70*, 41–40.
6. Plarre, R. Pheromones and other semiochemicals of stored product insects. A historical review, current application, and perspective needs. *Mitt. Biol. Bundesanst. Land-Forstwirtschaft.* **1998**, *342*, 13–83.
7. Barak, A.V.; Burkholder, W.E.; Faustini, D.L. Factors affecting the design of traps for stored-product insects. *J. Kans. Entomol. Soc.* **1990**, *63*, 466–485.
8. Wright, J.; Cogan, P. Trapping and sampling: a proposal for standards in reporting. *J. Stored Prod. Res.* **1995**, *31*, 175–176.
9. Subramanyam, B.; Hagstrum, D.W. Sampling. In *Integrated Management of Insects in Stored Products*; Subramanyam, B., Hagstrum, D.W., Eds.; Marcel Dekker: New York, 1996; 135–193.
10. Wileyto, E.P.; Ewens, W.J.; Mullen, M.A. Markov-recapture population estimates: a tool for improving interpretation of trapping experiments. *Ecology* **1994**, *75*, 1109–1117.
11. Trematerra, P. The use of sex pheromones to control *Ephestia kuehniella* zeller (Mediterranean flour moth) in Mills by Mass Trapping and Attracticide (lure and kill) Methods. In *Proc. 6th Intern. Working Conf. Stored-Product Prot., Canberra*; 1994; 375–382.
12. Buchelos, C.T.; Levinson, H.Z. Efficacy of multisurface traps and lasiotraps with and without pheromone addition, for monitoring and mass-trapping of *Lasioderma serricornis* F. (Col., Anobiidae) in insecticide-free tobacco stores. *J. Appl. Entomol.* **1993**, *116*, 440–448.
13. Pierce, L. Suppression of cigarette beetles, *Lasioderma serricornis* (Fabricius), by focused mass trapping. In *Proc. 7th Intern. Working Conf. Stored-Product Prot., Beijing*; 1998; 1455–1463.
14. Trematerra, P. The use of attracticide method to control *Ephestia kuehniella* Zeller in flour mills. *Anz. Schaedlingskd., Pflanzenschutz, Umweltschutz* **1995**, *68*, 69–73.
15. Vail, P.L.; Hoffmann, D.F.; Tebbets, J.S. Autodissemination of *Plodia interpunctella* (Hübner) (Lepidoptera: Pyralidae) granulosis virus by healthy adults. *J. Stored Prod. Res.* **1993**, *29*, 71–74.
16. Prevett, P.F.; Benton, F.P.; Hall, D.R.; Hodges, R.J.; Dos Santos Serodio, R. Suppression of mating in *Ephestia cautella* (Walker) (Lepidoptera: Phycitidae) using microencapsulated formulations of synthetic sex pheromone. *J. Stored Prod. Res.* **1989**, *25*, 147–154.
17. Levinson, H.Z. Integrated manipulation of storage pests involving insectistasis. *Mitt. Dtsch. Ges. Allg. Angew. Ent.* **1983**, *4*, 102–103.
18. Phillips, T.W.; Cogan, P.M.; Fadamiro, H.Y. Pheromones. In *Alternatives to Pesticides in Stored-Product IPM*; Subramanyam, B., Hagstrum, D.W., Eds.; Kluwer Academic Publishing: Norwell, 2000; 273–302.
19. Shumann, S.; Epsky, N.D. Computerized monitoring of stored-product insects populations. In *Proc. 7th Intern. Working Conf. Stored-Product Prot., Beijing*; 1998; 1429–1436.
20. Arbogast, R.T.; Weaver, D.K.; Kendra, P.E.; Brenner, R.J. Implications of stial distribution of insect populations in storage ecosystems. *Environ. Entomol.* **1998**, *27*, 202–216.

Physical Barriers: Vertebrate Pests

Carolyn J. Randall

Pesticide Education Program, Michigan State University, East Lansing, Michigan, U.S.A.

INTRODUCTION

Exclusion is an integrated pest management (IPM) technique that works by keeping the pests out. Physical barriers are one type of exclusion technique that prevents vertebrates from entering places where they are not wanted such as in or around buildings, landscape areas and gardens, and croplands.

Many people do not wish to see vertebrate pests such as birds, raccoons, squirrels, and chipmunks harmed. Physical barriers are considered a humane way of controlling them. Barriers generally do not kill or harm the pest and may be used to protect crops, people, and property from vertebrate pest damage. Physical barriers include such things as fences, wires, netting, and screens that are used to control vertebrate pests such as birds, rats, mice, squirrels, bats, skunks, raccoons, opossums, rabbits, and white-tailed deer.

PHYSICAL BARRIERS FOR BIRDS

The three main pest birds in urban areas in the United States are pigeons, European starlings, and house sparrows. In urban areas, the three main barrier methods used to prevent pest birds from roosting and nesting on buildings are netting, covers or ramps, and spikes.^[2,3]

- **Netting:** Netting is used to block access of birds to large roosting areas in structures. Netting is especially useful in warehouses and around mechanical equipment areas where aesthetics are of minor consideration. It has been used successfully on cooling towers. Plastic nets have replaced metal and fiber nets in bird control. Plastic nets are normally extruded black polypropylene and are made with an ultraviolet (UV) inhibitor to reduce UV degradation. Knotted nets are also available. Some newer designs in nets are less obtrusive and come in custom colors. Nets will last from 2 to 5 years or longer, depending on exposure to sunlight.
- **Covers or ramps:** Custom-designed covers for ledges, window air-conditioning units, and roof edges are the best technical solutions to keep birds from infesting these sites (Fig. 1). The high cost of this method usually eliminates this option on large buildings that have extensive roosting sites. But

covers are valid options where limited applications will keep birds off selected sites and where aesthetics are important. The covers usually consist of sheet metal installed at a 45° angle to prevent the birds from landing. Sheet metal, wood, Styrofoam blocks, stone, and other materials can be fastened to ledges to accomplish the desired angle.

- **Spikes:** Porcupine wire (Fig. 2), sharp metal spikes, or any similar “bed of nails” can stop birds from roosting on ledges. If aesthetics are important, these devices are usually limited to areas where they cannot be easily seen. Some newer products, such as clear plastic spikes, may be more aesthetically pleasing.

Several pest birds, including a variety of black birds, can become major pests of agricultural crops. Netting and monofilament lines are barrier methods used to control pest birds in agricultural areas.^[3] Netting (Fig. 3) is the most effective method for controlling bird damage in agricultural areas. However, there is a high labor cost for installation and removal of netting, and this method is usually cost-effective for only the most valuable crops. Monofilament lines (cords of fine wire) have been used to reduce bird damage at landfills, fish hatcheries, public parks, and agricultural fields. The lines are stretched across the areas in a variety of spacings and configurations (grid patterns, parallel patterns, etc.) and at varying heights, depending on the species to be controlled. Species responses have been quite variable. For example, gulls, crows, and sparrows appear to be particularly sensitive to lines and have been successfully repelled. To protect crops, lines are usually practical only for small plots or home gardens. Commercial plastic strips (Fig. 4) can provide bird-proof barriers for doors to warehouses, grain storage areas, and other buildings. These strips can be hung from the top of the doorframe to ground level, allowing easy access for people and equipment.

PHYSICAL BARRIERS FOR RODENTS

Pest rodents include domestic rats (Norway rats and roof rats) and house mice, and wild rodents such as voles, tree squirrels, chipmunks, and woodchucks.

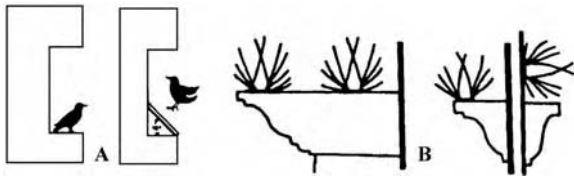


Fig. 1 A wooden, metal, or Plexiglas covering over a ledge at a 45° angle (A) or a porcupine wire (B) can be used to prevent roosting and nesting. (University of Nebraska Cooperative Extension Service.)

Rats and mice that are pests of structures can be kept out of buildings by rodent-proofing.^[1,2] This involves sealing cracks and holes in building foundations and exterior walls, blocking all openings around water and sewer pipes, and screening air vents. Doors should be caulked and sealed to ensure a tight fit, especially between the door and floor thresholds. Windows and screens should also fit tightly. For the building interior, spaces inside hollow block voids or behind the wallboard and broken blocks and holes around pipes should be sealed or repaired. In addition, gnaw holes can be repaired or stuffed with copper wool and floor drains equipped with sturdy metal grates held firmly in place.

The most common rat pest in the United States is the Norway rat (Fig. 5). Roof rats are found in certain parts of the country (mostly the coastal states). If roof rats are a problem, it is also important to block openings around electric lines, air vents, and telephone wires, and to caulk and close all openings on upper floors and the roof.^[2]

Keeping mice off the building will be more difficult than keeping off rats because mice are reported to be able to squeeze through an opening as little as 1/4 in. diameter.^[2] Chipmunks and other wild rodents occasionally enter buildings as well. One-quarter-inch metal mesh, caulking, or other appropriate materials

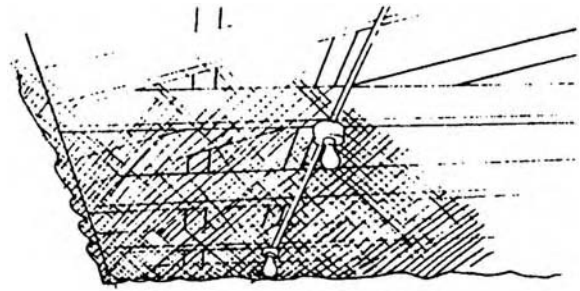


Fig. 3 Netting can be used to exclude birds from building rafters and fruit trees. (University of Nebraska Cooperative Extension Service.)

can be used to close openings where they could gain entry.

The first step in keeping squirrels out of buildings is to find out where they are entering. Common points of entry include damaged attic louvers, ventilators, soffits, joints of siding, knotholes, openings where utility wires or pipes enter, chimneys, and flashing. Squirrels may gnaw directly through the siding and shingles, too. To keep squirrels out, heavy-gauge 1/2-in. hardware cloth or sheet metal can be used to seal most openings.^[1,2]

Voles may cause extensive damage to orchards, ornamentals, and tree plantings by girdling seedlings and mature trees. Hardware cloth cylinders may be used to exclude voles from seedlings and young trees. Hardware cloth mesh that is 1/4 in. or less should be used and buried 6 in. to keep voles from burrowing under the cylinder.^[1,3] Hardware cloth may also be used to keep chipmunks out of flowerbeds. Seeds and bulbs can be covered by 1/4-in. hardware cloth and

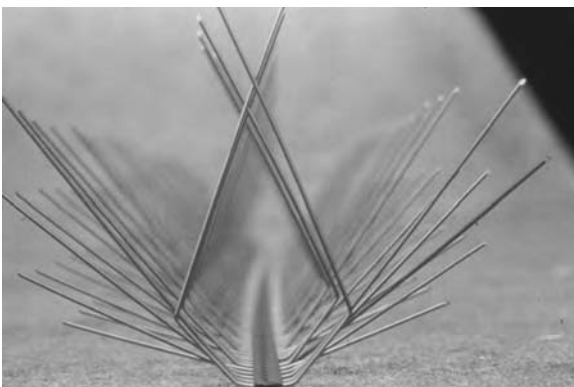


Fig. 2 Nixalite (porcupine wire). (U.S. Environmental Protection Agency.)

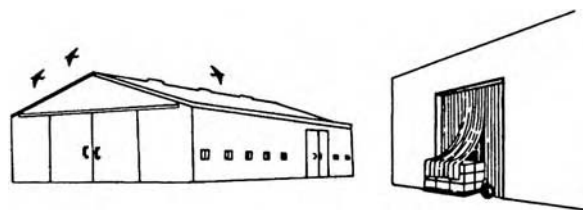


Fig. 4 Bird-proofing of buildings. (University of Nebraska Cooperative Extension Service.)



Fig. 5 Norway rat, *Rattus norvegicus*. (U.S. Environmental Protection Agency.)

the cloth covered with soil. The cloth should extend at least 1 ft past each edge of the planting.^[1,3]

Fences can be used to protect home gardens from woodchuck damage. Fences should be at least 3 ft high (woodchucks are good climbers) and made of heavy woven wire. To prevent burrowing under the fence, the lower edge should be buried 10–12 in. in the ground or bent at an L-shaped angle leading outward and buried in the ground by 1–2 in. An electric wire may be placed 4–5 in. off the ground and the same distance outside the fence to prevent climbing and burrowing. Sometimes the electric wiring alone is enough to discourage woodchucks from entering gardens. Bending the top 15 in. of the wire fence outward at a 45° angle will prevent climbing over the fence.

PHYSICAL BARRIERS FOR BATS

Bats' (Fig. 6) roosting and hibernating sites may occur in building attics, wall and ceiling voids, belfries, and

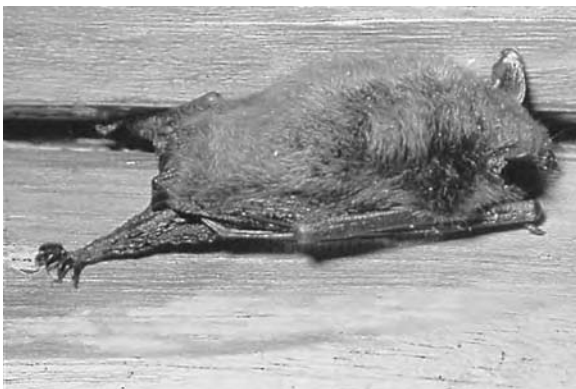


Fig. 6 Little brown bat, *Myotis lucifugus*. (U.S. Environmental Protection Agency.)

chimney voids. The peak months for bat complaints are June and July.^[2] Unfortunately, this is the worst time of year for control. At this time, bats are rearing young in their colonies. Bat-proofing during this period will trap the young bats inside. The best time of the year to bat-proof a building is either in late fall (after bats have left for hibernation) or in late winter and early spring (before the bats arrive). Bat-proofing a building involves sealing all but one or two principal openings and then waiting 3–4 days for the bats to adjust to using the remaining openings. Those openings should be sealed some evenings just after the bats have left for their nightly feeding. Bat valves can also be used. These devices are placed over the remaining openings and allow the bats to leave but not to return. The same materials used for rodent-proofing may be used to bat-proof buildings: 1/4-in. hardware cloth, screening, sheet metal, caulking, expanding polyurethane foam, steel wool, and duct tape. For older buildings with many openings, large sections of plastic bird netting can be draped over the roof areas to keep out bats at a reasonable cost.

PHYSICAL BARRIERS FOR SKUNKS, RACCOONS, AND OPOSSUMS

Skunks, raccoons, and opossums can be prevented from entering buildings by repairing breaks in foundations and screening crawlspace vents with hardware cloth.^[1,2] If the animal is currently living under the

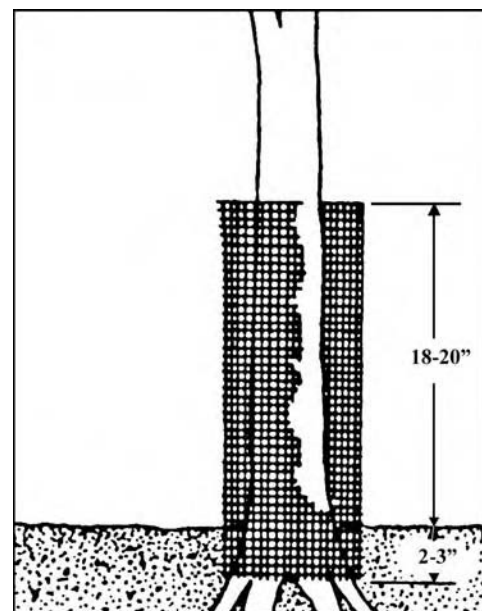


Fig. 7 A cylinder of hardware cloth or other wire mesh can protect trees from rabbit damage. (University of Nebraska Cooperative Extension Service.)

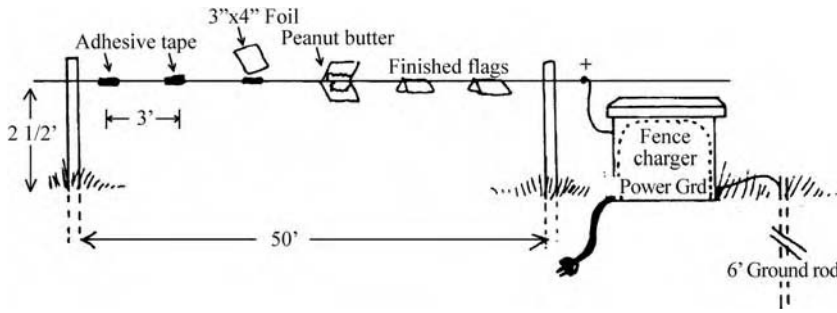


Fig. 8 The peanut butter fence: one type of temporary electric fence. (University of Nebraska Cooperative Extension Service.)

building, all openings but one should be sealed and a tracking patch of talc sprinkled at the opening. The area should be examined after dark and if the tracks show the animal has left, the last opening can be closed. To keep these animals out of attics, all openings should be sealed as for tree squirrels. Chimneys can be capped with a wire cage or other animal-proof covers.

PHYSICAL BARRIERS FOR RABBITS

One of the best ways to protect a backyard garden or berry patch from rabbits is to put up a fence. It does not have to be tall or especially sturdy. A fence of 2-ft chicken wire (1 in. or less mesh) with the bottom tight to the ground or buried a few inches is sufficient.^[3]

Cylinders of 1/4-in. mesh hardware cloth (Fig. 7) will protect valuable young trees or other landscape plants from gnawing rabbits. The cylinders should extend higher than a rabbit’s reach while standing on the expected snow depth, and stand 1–2 in. out from the trunk. Commercial tree guards or tree wraps are other alternatives.

A dome or cage of chicken wire secured over a small flowerbed will allow vulnerable plants such as tulips to get a good start before they are left unprotected.

PHYSICAL BARRIERS FOR WHITE-TAILED DEER

Fencing may be the only effective way to minimize deer damage especially in areas where the deer population is large and/or the crops are particularly valuable. Several fencing designs are available to meet specific needs.^[1,3] Temporary electric fences are a simple, effective way to protect garden and field crops during snow-free periods (Fig. 8). Permanent high-tensile electric fences provide year-round protection from deer and are best suited to high-value specialty or orchard crops. Permanent woven wire fences provide the ultimate deer barrier (Fig. 9). They require little maintenance but are very expensive to build.

Some factors to consider in determining what type of fence to build are the history of past deer numbers and extent of damage, deer pressure (i.e., the number of deer and their level of dependence on agricultural crops), value of the crop, and field size. With this information, a cost–benefit analysis should be prepared to determine the cost-effectiveness of fencing and the type of fence to install. Weigh the value of the crop to be protected against the acreage involved, costs of fence construction and maintenance, and life expectancy of the fence.

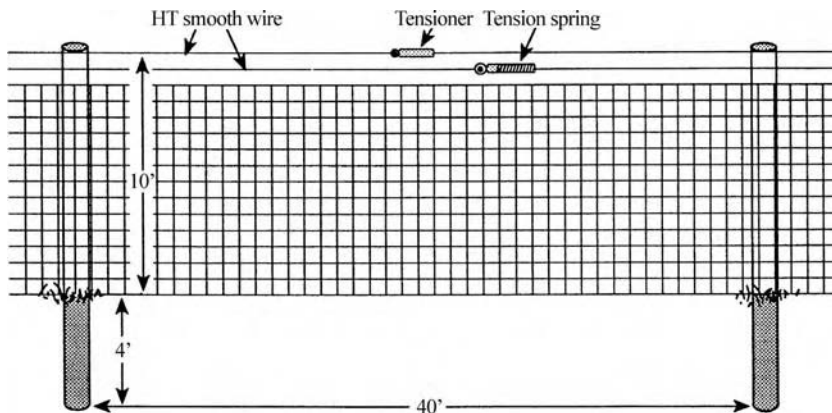


Fig. 9 The deer-proof woven wire fence. (University of Nebraska Cooperative Extension Service.)

CONCLUSION

Compared with some other vertebrate pest control methods, barrier methods have the advantages of being non-lethal, poison-free, and environmentally friendly. Other methods, such as hunting, baiting, and trapping animals, often require special licenses or permits. Because barrier methods do not kill or harm animals, most states allow them to be used without any special requirements. Thus property owners usually do not have to worry about breaking any state wildlife protection laws when using barrier methods. Overall, they provide safe, effective, and practical means for controlling vertebrate pests.

REFERENCES

1. Randall, C.J. *Vertebrate Pest Management, Extension Bulletin E-2050*; Michigan State University Extension, 1999; 1-73.
2. Wood, E.; Pinto, L. *Urban Integrated Pest Management: A Guide for Commercial Applicators. U.S. Environmental Protection Agency (735-B-92-001)*; DUAL and Associates: Arlington, VA, 1992. Module 3, Chapters 1-5.
3. Hygnstrom, S.E., Timm, R.M., Larson, G.E., Eds.; *Prevention and Control of Wildlife Damage*; University of Nebraska Cooperative Extension Service, USDA-APHIS, Great Plains Agricultural Council Wildlife Committee, 1994; Vol. II, D5-D40, D75-D80, E87-E120.

Phytosanitary Quarantine as a Pest Control Method

John S. Hartung

Plant Germplasm Quarantine Office, United States Department of Agriculture - Agricultural Research Service, Beltsville, Maryland, U.S.A.

INTRODUCTION

Agricultural crops are susceptible to a variety of pest threats, and it would be impossible to meet human needs for food and fiber without limiting losses to pests. The term “pest” includes insects and other invertebrates that attack crops, as well as weeds and pathogenic organisms.^[1] Losses to pests are limited by breeding for resistance to specific pests, modifying production practices to limit exposure to pests, and by applying agricultural chemicals.

This review will describe the basis and rationale used to establish and implement phytosanitary quarantines. Some examples and data will be drawn from the somewhat specialized phytosanitary quarantine procedures used to protect U.S. agriculture from exotic pests, while facilitating the importation of germplasm for plant breeding or conservation purposes.

BASIS FOR PHYTOSANITARY QUARANTINES

The ideal method of pest control is to avoid all exposure to the pest. This is possible in practice because many important pests have only local or regional, rather than global, distribution, often because of limitations on the ability of the pest to spread great distances without human assistance. Thus, if such an “exotic pest” does not occur in a particular region, it makes sense to take steps to prevent its human-assisted introduction.^[2] This was the rationale for the Plant Quarantine Act of 1912 (as amended), which provides the legal basis for regulatory actions in the United States to exclude pests that might be inadvertently imported on nursery stock, seeds, etc. Presumably, an effective strategy of pest exclusion would require the government to order the eradication of exotic pests should they be introduced and the prevention of interstate movement of such pests. These powers are provided in the United States by the Organic Act of 1944 and the Federal Pest Act of 1957, respectively. One hundred and six nations are signatories to the International Plant Protection Convention of 1951, which provides a harmonized framework for international plant-quarantine activities. In the United States, the responsibility for interpreting and implementing

these laws at the international level is assigned to the U.S. Department of Agriculture, Animal and Plant Health Inspection Service (USDA-APHIS). Each state also retains authority to implement analogous regulations directed at interstate movement of plant pests.^[2,3]

BENEFITS AND COSTS OF PHYTOSANITARY QUARANTINES

The concept of pest control by quarantine measures is both long established and widely practiced. Benefits of pest exclusion are real, easily understood, and widely shared. Both producers and consumers in the protected region benefit from reduced production costs when a pest is not present and, therefore, does not require control. The result is higher yield and improved quality of food and fiber products, with less adverse environmental impact from control measures. Producers also enjoy easier access to markets when pathogens/pests subject to phytosanitary quarantine in the target market do not occur in their region. However, producers in regions subjected to a phytosanitary quarantine must bear additional production and processing costs in order to attempt to gain access to international markets.

CHALLENGES FACING PHYTOSANITARY QUARANTINE PROGRAMS

Plant pest quarantines are conceptually simple but can be difficult to implement in practice. One challenge is to allow for the safe and efficient importation of germplasm or varieties intended for crop improvement without simultaneously importing damaging foreign pests. The problem in this case is that the host, with the assistance of human-assisted transport, can act as the vector of submicroscopic pathogens.^[4] Because none of the major agricultural crops grown in the United States are native to North America, plant breeding programs are dependent on foreign germplasm, which may come from regions of the world where pests targeted by quarantine regulations are known to occur. This is also true in other countries to a very large extent. Great concern has been expressed about the continuous “erosion” of genetic resources due to increasing human

population pressures that lead to loss of habitat for the wild ancestors of agricultural crops. This concern has led to a significant effort in the United States to import such germplasm for conservation.

Quarantine programs must define the pests of concern and the methods that will be used to prevent their introduction. These pests include a diverse range of insects as well as plant pathogenic fungi, bacteria, phytoplasmas, viruses, and viroids. A pest should be of quarantine significance only if it does not occur on a particular host species in the importing country, and if it has an ecological range in its native land similar to what is found in the importing country.^[2,3] Therefore, the national program must be effectively implemented and must have sufficient capacity to thoroughly test or inspect imported plant material for the presence of pests. There is a risk to the sustainability of agricultural production if the programs are inadequately designed or funded, and if they allow the introduction of pathogens/pests of quarantine significance. There is also a risk that if the quarantine program is too cumbersome, the development of improved horticultural or agronomic varieties will be unnecessarily delayed because of the unavailability of germplasm. Loss of genetic diversity in agricultural germplasm may result if efforts to conserve germplasm are impeded by limitations in the design, management, or funding of phytosanitary quarantine programs.

PROBLEMS IN IMPLEMENTING PLANT QUARANTINES

Eradication of newly observed infestations of pests is a necessary part of a successful disease control by an exclusion strategy, because introductions of exotic pests will inevitably occur.^[2] Decisions to proceed with eradications must be made rapidly and occasionally with imperfect information. The taxonomy of microorganisms, in particular, is an arcane and evolving field of science. This can result in misdirected eradication programs targeted at pests that have been, based on hindsight, misidentified. This occurred with a bacterial disease of citrus that was confused with citrus bacterial canker in Florida in 1984.^[5] Such mistakes can alienate growers and the general public towards quarantine efforts.

Unfortunately, phytosanitary quarantines can be misused as trade barriers. These can be based on taxonomic disputes or on the requirement by an importing country that there be “zero” risk of importation of a pathogen/pest with an agricultural commodity. Zero risk is, of course, impossible to achieve.

The intention of the phytosanitary quarantine is to prevent the spread of foreign pests by people acting as vectors by carrying infected or infested host plant

material. Pests that are easily disseminated by natural means are likely to spread despite human efforts until they reach a natural barrier and are less likely to be contained by quarantine measures. Therefore, the cooperation of an informed public is vital to the long-term success of this pest control strategy.^[2] For example, the public must declare and surrender produce upon arrival at international airports. Failure to do so risks civil penalties but, more importantly, the introduction of plant pests. Citizens must also be occasionally willing to allow the destruction of personal property (e.g. fruit trees) in the interest of a pest eradication program, as has been the case with the citrus canker eradication program in Florida since 1995. When the public does not understand either the biological basis of plant diseases or the consequences of insect infestations, they are likely to circumvent these regulations, which increases the risk for pest introduction.

EFFECTIVENESS OF PHYTOSANITARY QUARANTINE

The operation of the phytosanitary disease control system requires substantial investment. As one example, specially designed facilities and specially trained and dedicated personnel are required to perform the pathogen testing activities needed to facilitate safe importation of germplasm. The National Plant Germplasm Quarantine Center (NPGQC) is currently under construction at Beltsville, Maryland, and will cost about \$23 million when finished.^[6] In view of the expense, how can the effectiveness of this strategy be measured? This is impossible to answer rigorously, because we do not know how frequent pest introductions would have been in the absence of the existing phytosanitary quarantine programs. In spite of phytosanitary quarantines, pests of quarantine significance have been introduced into the United States in recent years, including citrus bacterial canker disease to Florida in 1986 and 1995,^[7] and plum-pox virus in Pennsylvania in 1999,^[8] both most likely as a result of smuggling of propagative materials. The Asian Long-Horned Beetle has also been introduced into the United States, presumably as larvae in wooden shipping pallets from China. In spite of these counter examples, the widespread international adoption and implementation of the concept of phytosanitary quarantines is an indication that these programs are perceived to be effective.^[4,9]

Eradication programs are necessary when quarantine barriers are breached. Vigorous efforts are underway to eradicate the localized introductions of plum-pox citrus canker disease and the Asian Long-Horned Beetle to prevent their long-term establishment. In the case of citrus canker, the fresh fruit portion

(20%) of Florida's \$8.5 billion citrus industry is at risk if the disease becomes established. In response, the state and federal governments appropriated \$215 million to fund current eradication efforts.^[7] This example, of only one host/pathogen combination, can give the reader an appreciation of the important role quarantine regulations play in protecting agriculture.

Historical data on pathogen interception in germplasm is available from the NPGQC of the USDA. One may infer from this data the relative risk of inadvertent pest importation with plant material in the absence of quarantine measures. Submicroscopic pathogens have been detected in about 50% of clonally propagated germplasm of apples, pears, potatoes, and sweet potatoes. Similarly, about 10% of the clonally propagated stone fruit germplasm was similarly infected, and plum-pox virus has been intercepted at the NPGQC three times since 1995. Thus, exotic pathogens are regularly intercepted by the testing program. However, the majority of interception events prevent the introduction of novel strains of pathogens that are already present in the United States. These pathogens are, however, a concern to the USDA, the industry, and various states. It should be noted that germplasm submitted to this testing program has been carefully selected by professionals and inspected for visible disease/pest symptoms prior to submitting it for testing. In the absence of this additional safeguard (i.e., smuggling), the rate of pest contamination would likely be much higher.

How can the effectiveness of quarantine programs be improved? Research on taxonomic matters is critical for the long-term success of these programs, as has been shown, e.g., in the case of citrus bacterial canker disease, where an eradication program was terminated after the target was shown to be a victim of misidentification.^[5] Research is also needed to develop faster and more sensitive diagnostic methods. In the final analysis, because people are the vectors for the pests that we are concerned with in phytosanitary quarantine, the importance of public education on

the threat posed by new pathogens and other pests cannot be overemphasized.^[2]

REFERENCES

1. Kahn, R.P. Biological concepts. In *Containment Facilities and Safeguards for Exotic Plant Pathogens and Pests*; Kahn, R.P., Mathur, S.B., Eds.; American Phytopathological Society: St. Paul, Minnesota, 1999; 8–16.
2. Kahn, R.P. Exclusion as a plant disease control strategy. *Annu. Rev. Phytopathol.* **1991**, *29*, 219–246.
3. Kahn, R.P. Regulatory concepts. In *Containment Facilities and Safeguards for Exotic Plant Pathogens and Pests*; Kahn, R.P., Mathur, S.B., Eds.; American Phytopathological Society: St. Paul, Minnesota, 1999; 3–7.
4. Kahn, R.P. The host as vector: exclusion as control. In *Pathogens, Vectors and Plant Diseases*; Harris, K.F., Maramorosch, K., Eds.; Academic Press: New York, 1982; 123–149.
5. Hartung, J.S.; Civerolo, E.L. Restriction fragment length polymorphisms distinguish *Xanthomonas campestris* strains isolated from Florida citrus nurseries from *Xanthomonas campestris* pv. *citri*. *Phytopathology* **1989**, *79*, 793–799.
6. Foster, J.A.; Claus, W.P. Planning and construction of the National Plant Germplasm Quarantine Center. In *Containment Facilities and Safeguards for Exotic Plant Pathogens and Pests*; Kahn, R.P., Mathur, S.B., Eds.; American Phytopathological Society: St. Paul, Minnesota, 1999; 93–107.
7. Gottwald, T.R.; Hughes, G.; Graham, J.H.; Sun, X.; Riley, T. The citrus canker epidemic in Florida: the scientific basis of regulatory eradication policy for an invasive species. *Phytopathology* **2001**, *91*, 30–34.
8. Levy, L.; Damsteegt, V.; Welliver, R. First report of plum pox virus (Sharka disease) in the United States. *Plant Dis.* **2000**, *84*, 202.
9. Kahn, R.P., Mathur, S.B., Eds.; *Containment Facilities and Safeguards for Exotic Plant Pathogens and Pests*; American Phytopathological Society: St. Paul, Minnesota, 1999; 213 pp.

Plant Breeding

Gurdev S. Khush

International Rice Research Institute, Davis, California, U.S.A.

INTRODUCTION

Worldwide, insect pests and diseases cause an estimated crop loss of 25% and thus compete with humankind for food and fiber. Genetic resistance to pests is the most preferred strategy for reducing crop losses because there is no cost to farmers, and resistant cultivars are easily adopted and disseminated unlike “knowledge-based technologies.” Moreover, concern for the environment is an important public concern issue these days, and management methods that minimize the use of crop protection chemicals are increasingly being favored.

Breeding for resistance started with the domestication of crop plants. The first plant breeders, those women and men who domesticated our crop plants, could save only those plants that did not suffer from pest depredation. They selected plants for pest resistance and changed the population structure of their crop species in favor of resistance genes. Domesticated species were grown in monoculture, which encouraged the evolution of pest populations capable of overcoming the resistance. Therefore plant breeding sets the stage for sequential cycles of pest resistance and pest susceptibility of crop plants. Several historical accounts document disastrous disease epidemics and insect outbreaks, which probably resulted from large plantings of pest-susceptible crops. For example, stem rust attack on U.S. wheat crop in 1917 destroyed more than 2 million bushels, forcing U.S. President Herbert Hoover to declare two wheatless days a week. Two million people died of the Bengal famine of India in 1942. Severe food shortage was caused by a disease epidemic on rice crops in the Bengal state.

Scientific breeding for pest resistance started in the early years of the 20th century after the discovery of Mendel’s laws of inheritance. Since then, numerous varieties of pest-resistant crops have been developed. Highly productive agriculture in developed countries is based on pest-resistant varieties. In the developing countries, host plant resistance received major attention during the last 40 years. Rice and wheat varieties with multiple resistance were in the vanguard of Green Revolution. For example, improved rice varieties developed at the International Rice Research Institute (IRRI) in the Philippines are resistant to as many as

four diseases and three insects (Table 1). Similarly, improved wheat varieties developed at the International Maize and Wheat Improvement Center (CIMMYT) in Mexico are resistant to stem rust, leaf rust, and yellow rust. A large-scale adoption of pest-resistant varieties has led to major increases in food production and has averted serious food shortages. For example, rice production worldwide increased by 120% from 257 million tons in 1966 to 572 million tons in 1997. Similar increases occurred in wheat and maize production.

GENETIC DIVERSITY FOR FOOD SECURITY

It is important to maintain diversity on farmers’ fields to reduce genetic vulnerability. For this purpose, breeders use diverse sources of resistance in developing resistant varieties. These include cultivated varieties, landraces, weedy relatives, or even closely related wild species. Crosses between elite germ plasm and related taxa can be routinely made, and gene transfer is easily accomplished. If the sources of resistance are not available within the primary gene pool, breeders resort to hybridization with distantly related wild species. Such crosses are difficult to make and breeders use special techniques such as embryo rescue and x-ray treatments.

Pest-resistant varieties with diverse traits, differing in growth duration and grain quality, and with tolerance to abiotic stresses are developed for diverse farming systems. For example, improved rice varieties differ in growth duration from 105 to 140 days (Table 1). Numerous parents are used for incorporating genes for multiple resistance, high-yield potential, good grain quality, and tolerance to abiotic stress. IR64, a widely grown rice variety, has 19 landraces and a wild species from eight countries in its ancestry (Fig. 1).

Resistant varieties, once introduced into on-farm production, do not remain resistant forever. Therefore varieties with diverse genes for resistance are required. For maintaining a continuous supply of pest-resistant varieties, it is important to consider the types of resistance available, the durability of resistance, and the breeding strategies.

Table 1 Growth duration and disease and insect reactions of rice varieties developed at IRRI

| Variety | Growth duration (days) | Disease or insect reactions | | | | | | | |
|---------|------------------------|-----------------------------|------------------|--------|--------------|------------------|-------------------|------------|------------|
| | | Blast | Bacterial blight | Tungro | Grassy stunt | Green leafhopper | Brown planthopper | Stem borer | Gall midge |
| IR5 | 140 | MR | S | S | S | R | S | MR | S |
| IR8 | 135 | S | S | S | S | R | S | S | S |
| IR20 | 130 | MR | R | S | S | R | S | MR | S |
| IR22 | 125 | S | R | S | S | S | S | S | S |
| IR24 | 125 | S | S | S | S | R | S | S | S |
| IR26 | 120 | MR | R | MR | S | R | R | MR | S |
| IR28 | 110 | R | R | R | R | R | R | MR | S |
| IR32 | 140 | MR | R | R | R | R | R | MR | R |
| IR36 | 115 | MR | R | R | R | R | R | MR | R |
| IR38 | 120 | MR | R | R | R | R | R | MR | R |
| IR42 | 135 | MR | R | R | R | R | R | MR | R |
| IR46 | 130 | MR | R | R | R | R | R | MR | R |
| IR50 | 110 | S | R | R | R | R | R | S | — |
| IR54 | 120 | MR | R | R | R | R | R | MR | — |
| IR58 | 105 | MR | R | R | R | R | R | S | — |
| IR60 | 115 | MR | R | R | R | R | R | MR | — |
| IR62 | 115 | MR | R | R | R | R | R | MR | — |
| IR64 | 115 | MR | R | R | R | R | R | MR | — |
| IR66 | 115 | MR | R | R | R | R | R | MR | — |
| IR68 | 120 | MR | R | R | R | R | R | MR | — |
| IR72 | 115 | MR | R | R | R | R | R | MR | — |
| IR74 | 130 | MR | MS | R | R | R | R | MR | — |

R = Resistant; MR = moderately resistant; S = susceptible; (—) = not known.

TYPES OF RESISTANCE

Two types of resistance are generally recognized: 1) monogenic or major gene resistance, which is also referred to as vertical resistance and 2) polygenic or quantitative resistance, which is also known as horizontal resistance.

Monogenic resistance may be dominant or recessive. This type of resistance shows a differential interaction with biotypes of insects and races of disease organisms. It shows a high level of resistance to some races or biotypes, but a complete susceptibility to others. Biotype-specific or race-specific resistance is another term used to describe such resistance. When monogenic resistance is used in breeding, segregating populations show discreet phenotypic segregation for resistance and susceptibility.

Polygenic resistance is generally of moderate level. Moreover, there is no differential interaction when cultivars with polygenic resistance are infested with different biotypes or races. It is also referred to as

biotype-non-specific or race-non-specific resistance. With this type of resistance, a continuous variation from susceptibility to resistance is observed in segregating populations. Generally, several genes, each with a small contribution to resistance, are involved. These genes are also referred to as quantitative trait loci (QTL). When tagged with molecular markers, it is possible to follow the segregation of individual QTL in breeding populations. It is not uncommon to find cultivars that have monogenes as well as QTL for resistance.

DURABILITY OF RESISTANCE

Durable resistance is defined as the resistance that remains effective while a cultivar possessing it is widely cultivated. It depends upon the type of resistance, the population structure, the evolutionary biology of the pathogen, and the interaction of crop management practices with host resistance.

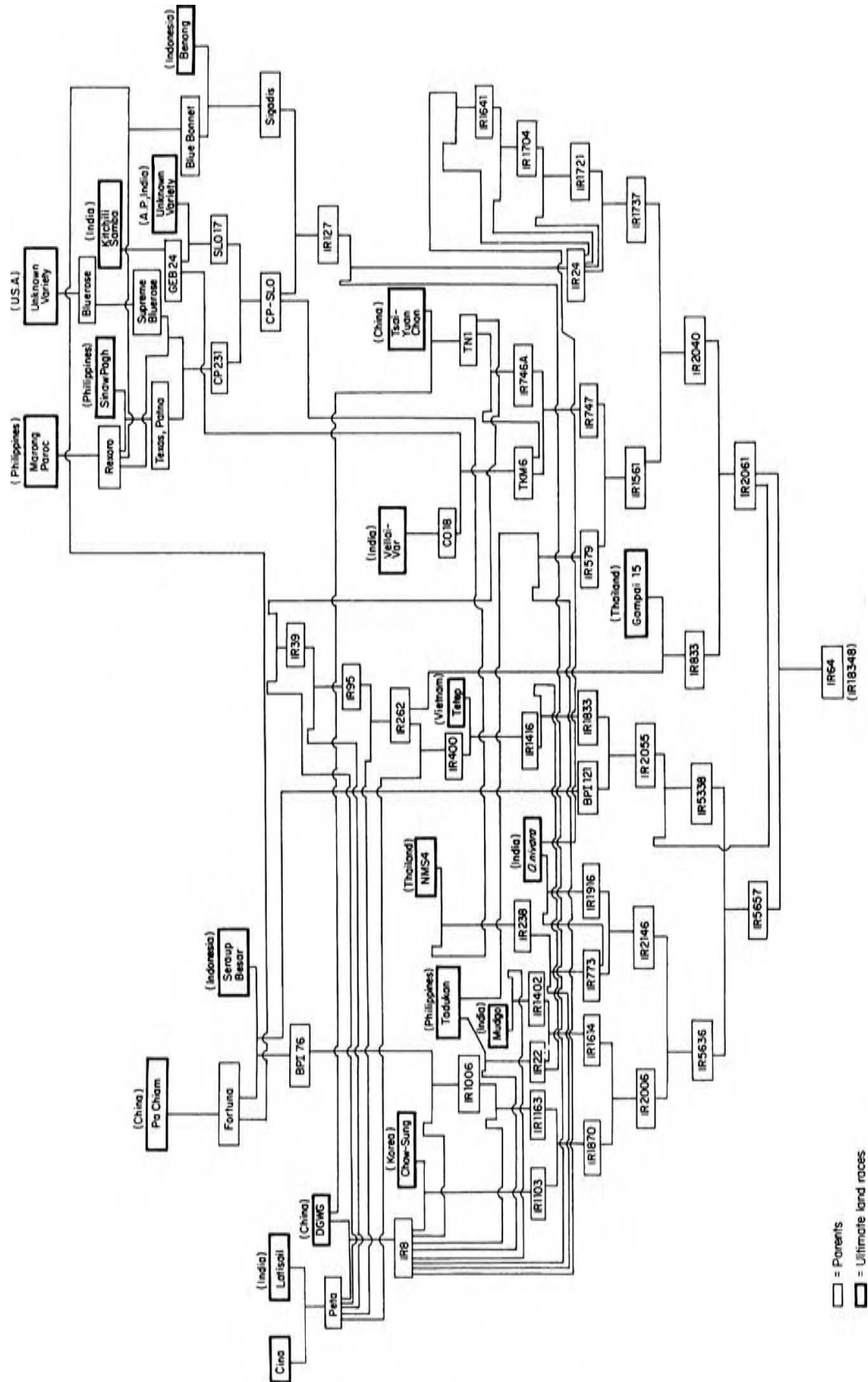


Fig. 1 Pedigree of multiple disease and insect-resistant rice variety IR64. Nineteen landrace and one wild species were used to develop this variety.

Horizontal resistance is generally considered more durable. Because the level of resistance is moderate, it does not exert too much selection pressure on the insect pest or the disease organism. Thus the chances of developing resistance-breaking biotypes or races are minimized. Monogenic resistance, on the other hand, is generally of high level and exerts pressure on the insect pests and pathogens to evolve and to overcome resistance. There are many examples of monogenic resistance breaking down in 6–10 years. However, there are several examples of monogenic resistance remaining stable for long periods. For example, wheat cultivars with monogenes SR2, SR24, and SR26 for rust resistance have been grown for more than 30 years and are still resistant. Coastal bermuda grass has a dominant gene for rust resistance. It has been grown for over 50 years and is still resistant. A major gene for resistance to the Milo disease of sorghum has held up for over 50 years. Tomato cultivars with resistance to as many as 14 diseases have been developed with monogenes, and the resistance has been durable. Polygenic resistance is not always stable either. For example, polygenic resistance to *Septoria tritici* has been eroding slowly.

BREEDING STRATEGIES

An appropriate breeding strategy for host plant resistance provides durable resistance. Obviously, polygenic or horizontal resistance is more durable; thus it is the preferred strategy. However, it is not always possible to breed for horizontal resistance. Sometimes, the donors with polygenic resistance are not available. Breeding for polygenic resistance is also laborious and a long-term undertaking. In the face of disease epidemics or insect outbreaks, breeders are expected to come up with resistant varieties in a short period. Under the circumstances, breeders turn to major genes for resistance. Four strategies are used for the utilization of major genes.

Sequential Release of Cultivars with Single Genes for Resistance

This strategy has been used in a wheat breeding program for Hessian fly resistance in the United States. A single gene for resistance is incorporated into a commercial variety, which is widely grown for several years. When biotypes virulent to this cultivar appear, another cultivar with a new gene for resistance is released. This strategy was also used to control stem rusts of wheat in Australia between 1938 and 1950, and to control brown planthoppers of rice in Asia between 1970 and 1990.

Pyramiding of Resistance Genes

This strategy aims to combine two or more major genes for resistance into the same variety. Several wheat varieties that combine up to five genes for resistance to stem rust have been developed in Australia. Canadian breeders adopted the same strategy for developing oats that are resistant to crown rust. Until recently, this approach depended upon the availability of disease races or insect biotypes capable of distinguishing between genotypes with various numbers of resistance genes. With the advent of molecular marker technology, the applicability of this approach has improved markedly. If the various major genes can be tagged with molecular markers, pyramiding can be accomplished by combining the closely linked molecular markers. In this molecular marker-aided pyramiding, an evaluation for resistance during the breeding process is not required. This approach was used to pyramid four genes for resistance to the bacterial blight of rice.

Development of Multiline Varieties

In this approach, isogenic lines for resistance are developed by backcrossing the donors with different genes for resistance with an elite, but susceptible, variety as recurrent parent. Six or seven backcrosses are required. Seeds of isogenic lines are mixed in equal proportion and released as a commercial multiline cultivar. If a component line of this cultivar becomes susceptible, it can be pulled out and replaced with another resistant line. This strategy was used to control crown rusts of oats in Iowa.

Geographical Deployment of Different Genes for Resistance

Varieties with different genes for resistance are recommended for different geographical regions of the country where crops cover a sizable area. This type of gene deployment is essentially a geographical multiline. This strategy was used for controlling leaf rusts of wheat in the United States.

It should be noted that all the abovementioned strategies for deploying major genes for resistance depend upon the availability of a number of different genes for resistance. Therefore a parallel program on a genetic analysis of resistant germ plasm to identify diverse genes for resistance is imperative.

CONCLUSION

It is obvious that for future food security, we must minimize the crop losses caused by diseases and insect

organisms. Various integrated pest management strategies have evolved, and host plant resistance is the basic component of most strategies. Conventional breeding has been successfully employed to develop crop cultivars with multiple resistance to diseases and insects. Recent breakthroughs in cellular and molecular biology have provided new tools for developing pest-resistant cultivars. Techniques of embryo rescue permit the hybridization and the transfer of genes for pest resistance from wild species to cultivated varieties and, thus help broaden the gene pool. Novel genes from unrelated plants, animals, and microorganisms can now be introduced through genetic engineering. For example, *Bt* gene from *Bacillus thuringiensis* has been introduced into several important crops, such as corn and cotton, and transgenic cultivars are highly resistant to corn borers and pink boll worm, respectively. Molecular genetic maps of many crop species have been prepared, and the major genes as well as QTL for disease resistance are being tagged with molecular markers. The efficiency of breeding methods is higher where molecular marker-aided selection can be applied, particularly for resistance traits of low heritability governed by QTL. With the application of molecular marker-aided selection, chances for developing crop varieties with durable resistance have been improved. Thus for continuous supply of pest-resistant crop varieties, breeders must utilize conventional as well as biotechnological approaches and

work with plant pathologists, entomologists, and biotechnologists.

BIBLIOGRAPHY

- Bonman, J.M.; Khush, G.S.; Nelson, R.J. Breeding rice for resistance to pests. *Annu. Rev. Phytopathol.* **1992**, *30*, 485–506.
- Khush, G.S. Disease and insect resistance in rice. *Adv. Agron.* **1977**, *29*, 265–341.
- Khush, G.S. Multiple disease and insect resistance for increased yield stability in rice. In *Progress in Irrigated Rice Research*; International Rice Research Institute: Manila, Philippines, 1989; 79–92, PO Box 933.
- Khush, G.S. Selecting Rice for simply inherited resistances. In *Plant Breeding in 1990s*; Stalker, H.S., Murphy, J.P., Eds.; CAB International, 1992; 303–323.
- Khush, G.S. Modern varieties—their real contribution to food supplies and equity. *GeoJournal* **1995**, *335*, 275–284.
- Khush, G.S.; Chaudhary, R.C. Role of resistant varieties in integrated pest management of rice. In *Extension Bulletin No. 162*; Food and Fertilizer Technology Center: Taipei City, Taiwan, 1981.
- Khush, G.S.; Brar, D.S. Genetics of resistance to insects in crop plants. *Adv. Agron.* **1991**, *45*, 223–274.
- Panda, N.; Khush, G.S. *Host Plant Resistance to Insects*; CAB International Wallingford: United Kingdom, 1995; International Rice Research Institute: PO Box 933, Manila, Philippines, 431 pp.

Plant Food to Enhance Performance of Natural Enemies in Mass Rearing and the Field

Felix L. Wäckers
P.C.J. van Rijn

*Center for Terrestrial Ecology, Netherlands Institute of Ecology (NIOO-KNAW),
Heteren, The Netherlands*

INTRODUCTION

Due to their ability to regulate the population density of arthropod herbivores, parasitoids and predators play an important role in biological control. Predators and parasitoids are usually identified by their carnivorous lifestyle. Due to this bias, we easily overlook the fact that the majority of these “carnivores” also require plant-derived foods as a source of nutrients. This vegetarian side of the menu may include various plant substrates, such as pollen, or nectar and other sugar sources (e.g., fruits and honeydew). Plant-provided foods can have a dramatic impact on longevity, fecundity, and distribution of predators and parasitoids. As each of these parameters affects the local number of carnivores, the availability of suitable plant-derived food can have a major impact on mass-rearing programs, as well as on herbivore–carnivore dynamics in the field.

IMPACT OF NECTAR AND POLLEN FEEDING ON CARNIVORE FITNESS

The level at which predators or parasitoids depend on primary consumption varies. Wäckers and van Rijn^[1] distinguish between “life-history omnivory,” “temporal omnivory,” and “permanent omnivory.” Life history omnivores include those natural enemies that are strictly dependent on plant-derived food during part of their life cycle, such as hoverflies and many parasitoids. Temporal and permanent omnivores supplement their carnivorous diet during part of their life (e.g., host-feeding parasitoids) and throughout their life cycle (e.g., predatory mites and ladybird beetles), respectively.

Parasitoids emerge with a limited supply of energy. At emergence, their energy reserves often cover no more than 48 hr of the parasitoid’s energetic requirements. Sugar feeding can increase a parasitoid’s life span considerably: up to 20-fold under laboratory conditions.^[2,3] This means that parasitoids that

fail to replenish their energy reserves through sugar feeding will suffer severe fitness consequences. Sugar feeding can benefit a parasitoid’s fecundity, not only through an increase in reproductive life span, but also through a positive effect on the rate of egg maturation.^[2]

Life history omnivores with a predatory larval phase (such as lacewings, gall midges, wasps, and ants) use nectar as an energy source in their adult phase as well, increasing their reproductive life span or their foraging range. Some of these life history omnivores also feed on pollen. In hoverflies and certain lacewings this protein-rich substance appears to be essential to maintain egg production.

Permanent omnivores (such as anthocorid bugs, ladybeetles, and predatory mites) often use both prey and plant-provided food (pollen and nectar) for survival and reproduction. This diet expansion allows them to extend the seasonal period of performance.

THE USE OF FOOD SUPPLEMENTS

Mass Rearing

The basic concept that the fitness of adult biological control agents can be dramatically enhanced through the simple provision of food supplements has been long engrained in mass rearing practice. To facilitate rearing, adult insects are commonly provided with pollen or sugar sources such as (diluted) honey, honeydew, sugar water, or fruits. The actual choice of the supplementary food source is usually based on criteria like convenience (availability, shelf-life), economy (cost), or compatibility with existing rearing methods. The relative suitability of food sources for the predator or parasitoid has received little attention. Those studies that have investigated food suitability show that substantial differences exist among different types of pollen^[4] as well as nectar and honeydew with regard to their chemical composition and nutritional value.^[5,6] Given this variation, the issue of food suitability should receive more attention.^[7]

Biological Control

Biological control workers have regularly suspected that the absence of pollen and/or sugar sources in agriculture could impose a serious constraint on the effectiveness of natural enemies in the field.^[8,9] Hocking^[9] pointed out that lack of food availability can also prevent introduced parasitoids from establishing in classical biological control programs. We still have few data on the nutritional status of natural enemies under field conditions,^[10,11] but recent studies indicate that natural enemies can indeed be food deprived in the absence of flowering vegetation.^[12] Thus, adding food sources to agro-ecosystems could be a simple and effective way to enhance the effectiveness of biological control programs. Three types of approaches have been proposed to alleviate the shortage of food in agricultural systems:

1. *Diversification of agro-ecosystems:* Food sources can be provided by enhancing plant diversity in agro-ecosystems, either through the use of non-crops in undergrowth or field margins^[13,14] or through mixed cropping with crops featuring flowers or extrafloral nectaries. However, not all plant-provided food is suitable as a food source for parasitoids and predators. Flowers may not be perceived by (some) natural enemies, or can be unattractive or even repellent.^[15] Other flowers may be attractive, but hide their pollination rewards within constricted floral structures that prevent those natural enemies with unspecialized mouthparts from exploiting these food sources. In more diverse systems there might be a further snake in the grass. Many herbivores are dedicated flower feeders as well. This drawback can be avoided by selecting flowers that cater to biological control agents, while being unsuitable for herbivores.^[16,17]
2. *Artificial food supplements:* An alternative to the use of (flowering) plants is the use of artificial food supplements such as food sprays.^[18] Food sprays typically consist of a carbohydrate solution in combination with a source of protein/amino acids. Insects that utilize honeydew as a food source may be especially adapted to exploit this “artificial honeydew.” Many studies have identified short-term increases in the numbers of natural enemies such as parasitoids, lady beetles, lacewings, and predatory bugs as a result of food sprays, although impacts on pest numbers have rarely been investigated.^[19] The fact that nutritional requirements of natural enemies often differ considerably from those of pest insects can be used to develop selective food sprays, i.e., food sprays that sustain biological

control agents without providing a nutritional benefit to the pest insect.^[3,20]

3. *Crop-provided food:* Some crops produce suitable food supplements themselves. Many crops flower during part of their growing period. In crops grown for their seeds or fruits (e.g., cereals, citrus, beans) this flowering period may coincide with the period when the plant is specifically vulnerable to herbivore attacks. Some crops, such as peppers and tomatoes, even flower during a large part of the growing season, thereby maintaining populations of predatory mites and anthocorid bugs that can effectively suppress thrips pests.^[21] Some crops provide nectar also outside the flowering period. These so-called “extrafloral nectaries” may be found on leaves, stems, or fruits. By producing extrafloral nectar, plants can attract carnivores and obtain their protective services.^[22] Extrafloral nectaries have evolved independently numerous times. This shows that during evolution, food supplements have proven to be a successful method to enhance biological control. The extrafloral nectar trait is also found in a number of crops and can be a useful element in biological pest control.^[23] Examples of



Fig. 1 Parasitoid *Cotesia glomerata* feeding on extrafloral nectar of faba bean (*Vicia faba*).

extrafloral nectar-producing crops include *Prunus* spp. (cherry, plum, peach, almond), cassava, faba bean, zucchini, pumpkin, cashew, and cotton (Fig. 1). The crop-produced nectar may suffice as a food source for predators and parasitoids. In other cases, there may be room for plant breeding to improve the timing, quantity, and quality of nectar production to better match the nutritional needs of biological control agents.

CONCLUSIONS

The use of non-prey food can be a simple and economic method both to optimize mass rearing of predators/parasitoids and to boost their effectiveness in biological control programs. Nectar and pollen sources vary substantially with regard to their suitability as insect food. To optimize the impact of food provision in biological control, feeding requirements of both natural enemies and herbivorous pests should be considered when selecting food supplements. Differences in food ecology between both groups can be exploited to develop selective food supplements that support natural enemies while minimizing nutritional benefits for pests.

REFERENCES

1. Wäckers, F.L.; van Rijn, P.C.J. Food for protection: an introduction. In *Plant-Provided Food for Carnivorous Insects: A Protective Mutualism and Its Applications*; Wäckers, F.L., van Rijn, P.C.J., Bruin, J., Eds.; Cambridge University Press: Cambridge, 2005.
2. Jervis, M.A.; Kidd, N.A.C.; Heimpel, G.E. Parasitoid adult feeding behaviour and biocontrol—a review. *Biocontrol News Inf.* **1996**, *17*, 11N–26N.
3. Wäckers, F.L. A comparison of nectar- and honeydew sugars with respect to their utilization by the hymenopteran parasitoid *Cotesia glomerata*. *J. Insect Physiol.* **2001**, *47*, 1077–1084.
4. van Rijn, P.C.J.; Tanigoshi, L.K. Pollen as food for the predatory mites *Iphiseius degenerans* and *Neoseiulus cucumeris* (Acari: Phytoseiidae): dietary range and life history. *Exp. Appl. Acarol.* **1999**, *23*, 785–802.
5. Wäckers, F.L. Do oligosaccharides reduce the suitability of honeydew for predators and parasitoids? A further facet to the function of insect-synthesized honeydew sugars. *Oikos* **2000**, *90*, 197–201.
6. Lee, J.C.; Heimpel, G.E.; Leibe, G.L. Comparing floral nectar and aphid honeydew diets on the longevity and nutrient levels of a parasitoid wasp. *Entomol. Exp. Appl.* **2004**, *111*, 189–199.
7. Wäckers, F.L. Suitability of (extra-) floral nectar, pollen and honeydew as insect food sources. In *Plant-Provided Food for Carnivorous Insects: A Protective Mutualism and Its Applications*; Wäckers, F.L., van Rijn, P.C.J., Bruin, J., Eds.; Cambridge University Press: Cambridge, 2005.
8. Illingworth, J.F. Natural enemies of sugar-cane beetles in Queensland. *Queensl. Bur. Sugar Exp. Stn. Div. Entomol. Bull.* **1921**, *13*, 1–47.
9. Hocking, H. The influence of food on longevity and oviposition in *Rhyssa persuasoria* (L) (Hymenoptera: Ichneumonidae). *J. Aust. Entomol. Soc.* **1966**, *6*, 83–88.
10. Casas, J.; Driessen, G.; Mandon, N.; Wielaard, S.; Desouhant, E.; Van Alphen, J.; Lapchin, L.; Rivero, A.; Christides, J.P.; Bernstein, C. Energy dynamics in a parasitoid foraging in the wild. *J. Anim. Ecol.* **2003**, *72*, 691–697.
11. Lee, J.C.; Heimpel, G.E. Nectar availability and parasitoid sugar feeding. *Proceedings of the 1st International Symposium on Biological Control of Arthropods*, Honolulu, HI, 2003, 220–225.
12. Wäckers, F.L.; Steppuhn, A. Characterizing nutritional state and food source use of parasitoids collected in fields with high and low nectar availability. *IOBC WPRS Bull.* **2003**, *26*, 203–208.
13. van Emden, H.F. The role of uncultivated land in the biology of crop pests and beneficial insects. *Sci. Hortic.* **1965**, *17*, 121–136.
14. Altieri, M.A.; Whitcomb, W.H. The potential use of weeds in manipulation of beneficial insects. *Hortic. Sci.* **1979**, *14*, 12–18.
15. Wäckers, F.L. Assessing the suitability of flowering herbs as parasitoid food sources: flower attractiveness and nectar accessibility. *Biol. Control* **2004**, *29*, 307–314.
16. Baggen, L.R.; Gurr, G.M.; Meats, A. Flowers in tri-trophic systems: mechanisms allowing selective exploitation by insect natural enemies for conservation biological control. *Entomol. Exp. Appl.* **1999**, *91*, 155–161.
17. Wäckers, F.L. Gustatory response by the Hymenopteran parasitoid *Cotesia glomerata* to a range of nectar and honeydew sugars. *J. Chem. Ecol.* **1999**, *25*, 2863–2877.
18. Hagen, K.S. Ecosystem analysis: plant cultivars (HPR), entomophagous species and food supplements. In *Interactions of Plant Resistance and Parasitoids and Predators of Insects*; Boethel, D.J., Eikenbary, R.D., Eds.; John Wiley and Sons: New York, 1986; 153–197.
19. Rogers, M.E.; Potter, D.A. Potential for sugar sprays and flowering plants to increase parasitism of white grubs (Coleoptera: Scarabaeidae) by Tiphid wasps (Hymenoptera: Tiphidae). *Environ. Entomol.* **2004**, *33*, 619–626.
20. Romeis, J.; Wäckers, F.L. Nutritional suitability of individual carbohydrates and amino acids for adult *Pieris brassicae*. *Physiol. Entomol.* **2002**, *27*, 148–156.
21. Van den Meiracker, R.A.F.; Ramakers, P.M.J. Biological control of the western flower thrips *Frankliniella occidentalis*, in sweet pepper with the anthocorid predator *Orius insidiosus*. *Meded. Fac. Landbouwwetensch. Rijksuniversiteit Gent* **1991**, *56* (2a), 241–249.
22. Turlings, T.C.J.; Wäckers, F.L. Recruitment of predators and parasitoids by herbivore-injured plants. In *Advances in Chemical Ecology of Insects*; Cardé, R.T., Millar, J., Eds.; 2004; 21–75.
23. Rogers, C.E. Extrafloral nectar: entomological implications. *Bull. ESA* **1985**, *31*, 15–20.

Poison Baits

Gilbert Proulx

*Alpha Wildlife Research and Management Ltd., Sherwood Park,
Alberta, Canada*

INTRODUCTION

Poison baits attract and kill or impair the health of animals touching or consuming them. They are used mainly for the control of arthropods and rodents. This entry reviews the components of poison baits, the factors that impact on their efficacy, and their advantages and limitations.

POISON BAITS FOR ARTHROPODS

Poison baits are commonly used for the control of social insects, mites and ticks, flies, moths, bollworms, and crickets.

Composition

Poison baits are composed of a carrier, a toxicant, and an attractant.^[1] The carrier may be dry or liquid. It can be food (sugar, oils, meat) or inedible material (plastic, cloth). The carrier's longevity and resistance to environmental conditions may be enhanced with preservatives and binding material, or through encapsulation. The toxicant is usually a contact or stomach poison belonging to organophosphates, carbamates, or pyrethroids.^[2] The attractant may be an integral part of the carrier or a pheromone that is specific to one or many pests.

Efficacy

The efficacy of poison baits depends on the attraction of the carrier and the performance of the toxicant, and is linked to seasonal variations in weather conditions, population densities, and food-type preference. Yearly life cycles follow such variations and must be taken into consideration when deploying poison baits. When poison baits are properly used, reductions of pest populations may be significant (Table 1).

Advantages and Limitations

Contrary to conventional applications, poison baits have a limited impact on beneficial predatory insects and, because they are applied in specific areas, risk of environmental contamination and costs are lower.^[3] When used with slow-acting toxicants, poison baits may be used to destroy colonies of social insects that bring food back to their nest.^[4] Poison baits are useful in preventive programs to protect areas cleared of a pest from reinvasion from adjacent infested areas.^[5] When applied over large areas, poison baits must be easily deployed and must remain effective for some time. The effect of time on baits' performance may be counteracted by properly timing the deployment of baits to coincide with the emergence of target animals. Such timing is important to hamper insect resistance to pesticides.^[6]

POISON BAITS FOR RODENTS

Poison baits have been developed mainly for the control of mice and rats, but also for other rodents impacting on agriculture and forestry practices.

Composition and Distribution

The carrier may change seasonally for the same species, and differ from one species to another. Perishable baits (fruits, vegetables) are more efficacious, but their preparation, storage, and application pose problems. Grain baits and extruded pellets are the most popular carriers. Paraffin blocks are also used to improve the persistence of baits and increase their selectivity for gnawing rodents.^[7] Additives (e.g., fatty substances, carbon disulfide) may be used to improve acceptance and palatability of baits. However, in food warehouses and grain silos, sweetened-liquid bait stations may be

Table 1 Efficacy of various poison baits for arthropods

| Poison bait | Arthropod | Efficacy (percentage of reduction) | Reference |
|--|--|--|-----------|
| 52% glue, 32% granulated sugar, 14% dried blood, and 2% dichlorvos | Screwworm, <i>Cochliomyia hominivorax</i> | 75–80 | [12] |
| Pheromone-impregnated tags with deltamethrin | Bont tick, <i>Amblyomma variegatum</i> | 80 | [13] |
| 0.5% boric acid in 25% sucrose water bait | Argentine ant, <i>Linepithema humile</i> | 80 | [4] |
| Fipronil-fed nymphs | German cockroach, <i>Blattella germanica</i> | 100 | [14] |
| B-cyfluthrin or deltamethrin on odor-baited cloths | Testse, <i>Glossina pallidipes</i> | >90 | [5] |
| Malathion flour granules | Corn earworm, <i>Helicoverpa zea</i> | Equivalent to reduction obtained with 5 sprays | [3] |

more appropriate than solid food baits.^[8] Dyes to repel birds and distinguish toxic baits from food and feed, and emetics for the protection of pets and humans may be added to poison baits.^[7]

Baits can be treated with acute or chronic toxicants.^[9] Acute single-dose baits commonly used for the control of rats or mice are ANTU, crimidine, norbormide, pyrinuron, red squill, and reserpine. Less specific toxicants are bromethalin, cholecalciferol, sodium fluoroacetate and strychnine (the latter two are restricted in some jurisdictions), and zinc phosphide. Chronic baits consist of anticoagulant-type rodenticides such as brodifacoum, bromadiolone, chlorophacinone, difethialone, diphacinone, pindone, and warfarin.

The selectivity and attraction of poison baits are greatly influenced by bait placement. Baits located along foraging trails or, in the case of fossorial rodents, in burrow systems, are better accepted than those that are randomly broadcasted. Bait stations or special packaging (e.g., cellophane or plastic packets) are useful to minimize exposure of non-target animals to rodenticides.^[7]

Efficacy

The efficacy of poison baits varies greatly among populations and species, and may depend on bait formulations and distribution, poison concentrations, and time of year (Table 2). During dispersing or breeding seasons, food shortage periods, or harsh weather periods, animals may be more vulnerable to poison baits. When populations have a prevalence of animals that are either bait-shy or resistant to a rodenticide, control levels may drop considerably (Table 2).

Advantages and Limitations

Acute toxicants are useful for rapid population reduction. However, because symptoms of poisoning occur shortly after ingestion, animals may ingest sublethal doses and become bait-shy, or develop some tolerance. Consequently, acute toxicants should not be used more than once or twice per year.^[9] While some acute toxicants are effective for commensal rodents only, many others are hazardous for various non-target species, humans included, and have no antidote.

Anticoagulants represent over 95% of all poison baits used today. Their main advantage is that they do not induce “bait shyness.” When symptoms of toxicosis develop, animals have already consumed a lethal dose. Anticoagulants are safer than acute toxicants, and have low secondary hazards. However, they are effective only after multiple feedings. Where alternative food is available, animals may not ingest enough toxicant to die. Furthermore, enough antidote vitamin K may be present in rodents’ regular diet to counteract the pathological changes caused by anticoagulants. In agricultural fields, it is recommended to use anticoagulants before green-up.^[10,11]

FUTURE NEEDS

Poison baits are advantageous because they allow for the selective removal of pests. However, continuing research for more selective and effective poisons, training, and extension will be required in the future to efficiently use them, and to deal with the growing problem of resistance in pests.

Table 2 Efficacy of various poison baits for rodents

| Poison bait | Application | Rodent | Efficacy (percentage of population reduction) | Reference |
|--|----------------------------|--|---|-----------|
| Zinc phosphide | | | | |
| 2%—pelleted bait | Aerial broadcast | Roof rat, <i>Rattus rattus</i> | 18 | [15] |
| —grain bait | Prebaiting and hand baited | Richardson's ground squirrel (<i>Spermophilus richardsonii</i>) | May–June—60 July–August—95 | [16] |
| | Hand baited | Northern pocket gopher (<i>Thomomys talpoides</i>) | Summer—13 Early fall—17 | [17] |
| 0.8%—cabbage bait | Hand broadcast | Townsend ground squirrel (<i>Spermophilus townsendi</i>) | 39 | [18] |
| Strychnine | | | | |
| 0.40%—oats | Hand baited | Northern pocket gopher | Reproduction—17 Summer—36 Early fall—11 | [17] |
| 0.35%–0.44%—grain baits | Hand baited | Richardson's ground squirrel | 94 | [19] |
| | Mechanical broadcast | | 73–97 | |
| Cholecalciferol | | | | |
| 0.075%—wheat pellets | Bait station | House mouse, <i>Mus domesticus</i> | Early summer—85.0 Late summer–fall—33.0 Winter—92.0 | [20] |
| 0.075%—whole grain wheat | | | Early summer—52.0 Late summer–fall—5.0 Winter—72.0 | |
| Anticoagulants | | | | |
| 0.01% diphacinone oats | Mechanical broadcast | Deer mouse, <i>Peromyscus maniculatus</i> | May and September—100 | [21] |
| 0.005% chlorophacinone pellets and 0.005% bromadiolone pellets | Mechanical broadcast | Columbian ground squirrel (<i>Spermophilus columbianus</i>) | May—70–80 July—100 (before aestivation) | [22] |
| 0.005% brodifacoum pellets | Aerial broadcast | House mouse | 99 | [23] |
| 0.005% difenacoum oatmeal bait | Broadcast | Difenacoum-resistant Norway rat (<i>Rattus norvegicus</i>) populations | 25–89 | [24] |

REFERENCES

- Hill, D.S. *Agricultural Insect Pests of the Tropics and Their Control*; Cambridge Univ. Press: Cambridge, 1983.
- Dent, D. *Insect Pest Management*; CAB International: Oxon, 1991.
- Dowd, P.F.; Behle, R.W.; McGuire, M.R.; Nelsen, T.C.; Shasha, B.S.; Simmons, F.W.; Vega, F.E. Adherent malathion flour granules as an environmentally selective control for chewing insect pests of dent corn ears: insect control. *J. Econ. Entomol.* **1998**, *91* (5), 1058–1066.
- Klotz, J.; Greenberg, L.; Venn, E.C. Liquid boric acid bait for control of the Argentine ant (Hymenoptera: Formicidae). *J. Econ. Entomol.* **1998**, *91* (4), 910–914.
- Mangwiro, T.N.C.; Torr, S.J.; Cox, J.R.; Holloway, M.T.P. The efficacy of various pyrethroid insecticides for use on odour-baited targets to control tsetse. *Med. Vet. Med.* **1999**, *13*, 315–323.
- Smith, K.E.; Wall, R. Suppression of the blowfly *Lucilla sericata* using odour-baited triflumuron-impregnated targets. *Med. Vet. Entomol.* **1998**, *12*, 430–437.
- Marsh, R.E. Techniques used in rodent control to safeguard non-target wildlife. In *Transactions of the Western Section of the Wildlife Society Annual Meeting*; Laudenslayer, W.F., Jr., Ed.; Monterey, California, 1985; 47–55.

8. Shumake, S.A.; Gaddis, S.E.; Crane, K.A. Liquid baits for rodent control: a comparison of wild Norway versus wild ricefield rat response to glucose plus saccharine solutions. *Crop Prot.* **1997**, *16* (1), 35–39.
9. Savarie, P.J. The nature, modes of action, and toxicity of rodenticides. In *CRC Handbook of Pest Management in Agriculture*; Pimentel, D., Ed.; CRC Press Inc.: Boca Raton, Florida, 1981; Vol. III, 113–127.
10. Berdoy, M.; Smith, P. Arms race and rat race: adaptations against poisoning in the brown rat. *Rev. Ecol. (Terre Vie)* **1993**, *48*, 215–228.
11. Ali, S., Ed.; *Crop Protection with Chemicals*; Alberta Agriculture, Food and Rural Development: Edmonton, Alberta, 1997.
12. Coppedge, J.R.; Broce, A.B.; Tannahill, F.H.; Goodenough, J.L.; Snow, J.W.; Crystal, M.M. Development of a bait system for suppression of adult screwworms. *J. Econ. Entomol.* **1978**, *71* (3), 483–486.
13. Allan, S.A.; Barré, N.; Sonenshine, D.E.; Burrige, M.J. Efficacy of tags impregnated with pheromone and acaricide for control of *Amblyomma variegatum*. *Med. Vet. Entomol.* **1998**, *12*, 141–150.
14. Gahlhoff, J.E., Jr.; Miller, D.M.; Koehler, P.G. Secondary kill of adult male German cockroaches (Dictyoptera: Blattellidae) via cannibalism of nymph fed toxic baits. *J. Econ. Entomol.* **1999**, *92* (5), 1133–1137.
15. Lefebvre, L.W.; Holler, N.R.; Decker, D.G. Efficacy of aerial application of a 2% zinc phosphide bait on roof rats in sugarcane. *Wildl. Soc. Bull.* **1985**, *13* (3), 324–327.
16. Matschke, G.H.; Fagerstone, K.A.; Halstead, N.D.; LaVoie, G.K.; Otis, D.L. Population reduction of Richardson's ground squirrels with zinc phosphide. *J. Wildl. Manage.* **1982**, *46* (3), 671–677.
17. Proulx, G. Evaluation of strychnine and zinc phosphide baits to control Northern pocket gophers (*Thomomys talpoides*) in alfalfa fields in Alberta, Canada. *Crop Prot.* **1998**, *17* (2), 135–138.
18. O'Brien, J. Chopped cabbage baits for ground squirrel control in Nevada. In *Proceedings Vertebrate Pest Conference*; Howard, W.E., Marsh, R.E., Eds.; University of California: Davis, California, 1978; Vol. 8, 25–27.
19. Record, C.R. Ground squirrel and prairie dog control in Montana. In *Proceedings Vertebrate Pest Conference*; Howard, W.E., Marsh, R.E., Eds.; University of California: Davis, California, 1978; Vol. 8, 93–97.
20. Twigg, L.E.; Kay, B.J. Evaluation of Quintox[®] for control of feral house mice. *J. Wildl. Manage.* **1992**, *56* (1), 174–185.
21. Howard, W.E.; Marsh, R.E.; Cole, R.E. A diphacinone bait for deer mouse control. *J. For.* **1970**, *68* (4), 220–222.
22. Askam, L.R. Effectiveness of two anticoagulant rodenticides (chlorophacinone and bromadiolone) for Columbian ground squirrel (*Spermophilus columbianus*) control in eastern Washington. *Crop Prot.* **1985**, *4* (3), 365–371.
23. Brown, P.R.; Singleton, G.R. Efficacy of brodifacoum to control house mice, *Mus domesticus*, in wheat crops in Southern Australia. *Crop Prot.* **1998**, *17* (4), 345–352.
24. Cowan, D.; Dunsford, G.; Gill, E.; Jones, A.; Kerins, G.; MacNicoll, A.; Quy, R. The impact of resistance on the use of second-generation anticoagulants against rats on farms in Southern England. *Pestic. Sci.* **1995**, *43*, 83–93.

Poisonous Arthropods

Findlay E. Russell

Department of Pharmacology and Toxicology, University of Arizona, Tucson, Arizona, U.S.A.

INTRODUCTION

The phylum Arthropoda is said to contain more species of animals than in all the other animal phyla combined. Fortunately, only a small number can be considered sufficiently “venomous” or “poisonous” to be dangerous to other animals and humans. There are, however, many species especially among the spiders and the scorpions, whose venoms are toxic and even lethal to other invertebrates and vertebrates. In agriculture and the storage of food stuff, these arthropods can become pests and can be a potential danger to humans.

In this article, we are limited to those arthropods commonly found in the United States. We also need to exclude those arthropods whose bites or stings are not venomous, but the trauma of their bite or sting can elicit pain. We must also exclude those creatures that are vectors for certain bacterial, viral, or rickettsial diseases and those arthropods whose bites or stings give rise to allergic reactions.

ARACHNIDA

The arachnids (Fig. 1) consist of the scorpions, spiders, whipscorpions, solpugids, mites, ticks, and crustaceans. With respect to the spiders and the scorpions, their stings or bites, such as the bites of reptiles, do not necessarily result in envenomation. The author has witnessed bites by the spider *Latrodectus* sp. and stings by the scorpion, *Vejovis russelli* on humans that did not end in envenomation, and this phenomenon could probably be said for other venomous arthropods.

Scorpions

The scorpions are said to be the oldest known living terrestrial arthropods. There are at least a thousand species of which more than 50 worldwide are of a serious danger to humans. Scorpions spend the daylight hours under ground cover or in burrows. They emerge at night to ambush other arthropods or even small rodents, capture them with their pincers, sting and paralyze them, or tear them apart and digest their body fluids. They are also cannibalistic, the larger ones often feeding on the smaller. Scorpions live from 2 to 10 years, although there are reports of a 25-year life span.

Many scorpion venoms contain low molecular weight proteins, peptides, amino acids, nucleotides, and salts, among other components. The neurotoxic components are generally classified on the basis of their molecular size. The short-chain toxins are composed of 30 to 40 amino acid residues with three or four disulfide bonds and appear to affect potassium or chloride channels; while the long-chain toxins have 60 to 70 amino acid residues with four disulfide bonds and affect mainly the sodium channels. These particular toxins, often called “neurotoxins,” may have an effect on both voltage-dependent channels. There appears to be a high degree of cysteines in most of these venoms. The toxins can selectively bind to a specific channel of cells, thus impairing the initial depolarization of the action potential which results in their neurotoxicity. Not all scorpions, however, have venom fractions that affect neuromuscular transmission. The venoms may be deleterious to other arthropods, but may exert no significant systemic effects on humans.

The symptoms and signs of scorpion envenomation differ considerably, depending on the species. In the United States, the most common offenders are members of the family Vejovidae, generally found in the southwest and western states, as well as in Mexico, Central American, and South America. Their sting gives rise to localized pain, swelling, tenderness, and mild paresthesia. Systemic reactions are rare although weakness, fever, and muscle fasciculations have been reported. These same findings have been described for the stings of the giant hairy scorpion, *Hadrurus*, another Vejovidae. Envenomations by some members of the genus *Centruroides*, however, are clinically most important, particularly in the western United States, where *C. exilicauda* is found. Pain, followed by numbness or tingling over the involved part are common, and in children may give rise to restlessness, hypertonicity, abnormal and random head, neck, and eye movements, and opisthotonus; while in adults, tachycardia, hypertension, increased ventilation, weakness and motor disturbances may predominate. Respiratory difficulties may occur accompanied by excessive salivation. Treatment consists of bed rest, supportive drugs, including respiratory stimulants when needed, and diazepam. An antivenom produced by Arizona State University for *C. exilicauda* stings is available and approved by the State, but does not have the Food



Fig. 1 ARACHNIDA.

and Drug Administration (FDA) approval. A polyvalent antivenom is produced in Mexico. Recently, the continuous infusion of midazolam HCl has been used with indifferent success in serious *C. exilicauda* cases in Arizona.

Spiders

The genera of spiders that have been shown to produce significant bites on humans, some 40 species, have been noted elsewhere. Spiders have a hardened, stiff integument that encloses and supports their soft internal structures. Their organ systems are somewhat analogous to those of the vertebrates. Their body is divided into the cephalothorax and abdomen. They have a pair of pedipalpi, four pairs of walking legs, and most have spinning organs or spinnerets: fingerlike appendages usually located on the lower abdomen posteriorly. They are predaceous creatures, living on the body juices of live animals. Some live for less than a year, but some may survive over 20 years.

Of at least 200 species of spiders that have been implicated in significant bites on humans, approximately 25 are found in the United States. The two most important genera are *Latrodectus*, the widow spiders, of which there are five species, and *Loxosceles*, the brown or violin spider, there being 12 species. *Latrodectus mactans* and *Latrodectus hesperus* are the most frequently involved widow spiders, while *Loxosceles reclusa*, *Loxosceles arizonica*, and *Loxosceles deserta* account for the most frequent violin spider bites.

Widow spiders (*Latrodectus*)

Although both male and female widow spiders of the five species are venomous, only the female has fangs large and strong enough to penetrate the human skin. Most mature female widow spiders range in body length from 10 to 18 mm. The morphology and ultrastructure of the secretory gland of *L. mactans* was first described in 1967. More recently, a study has been reported on the chemistry of the venom gland of *Loxosceles intermedia*. The venom glands in the cephalothorax are joined by two ducts that lead to paired chelicerae. Using light microscopy and transmission

electron microscopy, the authors described two layers of striated muscle fibers with an extracellular matrix, a basement membrane structure, and a fibrillar collagen matrix separating the muscle zone from the secretory epithelial cells of holocrine type.

Those toxins of *Latrodectus* venom that have neurogenic effects are composed of polypeptides and large molecular weight proteins. The small polypeptide toxins interact with cation channels and display spatial structure homology, affecting the function of calcium, potassium, and sodium channels. A family of high molecular weight toxic proteins, 125 kDa, known as latrotoxins, are proteins of about 1000 amino acid residues, share a high degree of structural identity, and cause a massive presynaptic transmitter release from a diversity of nerve endings in vertebrates.

Bites by the black widow are described as pinprick-like, followed by a dull, occasionally numbing pain in the affected extremity, and by pain and cramps in one or several of the large muscle masses. Rarely is there any local skin reaction except during the first 60 min following the bite, but pileorection in the bite area is sometimes seen. Muscle fasciculations frequently occur within 30 min of the bite. Sweating is common, and the patient may complain of weakness and pain in the regional lymph nodes, which are often tender on palpation and are occasionally enlarged; lymphadenitis is frequently observed. Pain in the lower back, thighs, and abdomen is a common complaint, and rigidity of the abdominal muscles is seen in most cases in which envenomation has been severe. Severe paroxysmal muscle cramps may occur, and arthralgia has been reported. Hypertension is a common finding after moderate to severe envenomations in the elderly. Blood studies are usually normal. Deaths in the United States have been rare.

There is no effective first-aid treatment. In most cases, intravenous calcium gluconate will relieve muscle pain, but this may need to be repeated at 4- to 6-hour intervals for optimum results. Muscle relaxants, such as methocarbamol or diazepam, may be of value. Acute hypertensive crises may require intravenous nitroprusside. The use of antivenom, *L. mactans*, should be restricted to the more severe cases and when other measures have proved unsuccessful. It must be

used with caution. One ampule intravenously is usually sufficient. In patients who are under 16 or over 60 years or have any history of hypertension or hypertensive heart disease and who show significant symptoms and signs, the use of antivenom seems warranted; it also is appropriate in cases involving pregnancy.

Violin or brown spider (*Loxosceles*)

These spiders, sometimes misnamed “brown recluse” (one species), are common in Africa and the Americas. Gertsch notes more than 50 species with 11 for the United States although he related to the author at least two additional species.

These spiders are brown or tan in color and distinguished by the violin-shaped darker marking on the cephalothorax and their three pair of eyes. They vary in size somewhat related to the species; however, in general, adults measure from 6 to over 16 mm in length with legs of 8 to 35 mm. Both male and female are venomous. Although they are generally found under ground cover, rocks, and debris in the wild, they have invaded human habitats. For example, in Sierra Madre, CA, the author once found almost 200 *Loxosceles laeta* under the stage of a theater about 8 months after a visiting dance team from South America unpacked and staged their performance there.

The venom of *Loxosceles* sp. contains a number of enzymes of which a 32 kDa fraction, known as sphingomyelinase D, may be synergized with other venom components of similar molecular weight, or a metallo-proteinase of 32–35 kDa (loxolysin B), which also appears to be involved with the dermonecrosis and hemorrhage effects of the venom. One of the difficulties in determining the specific chemical nature of the toxins may be the different techniques used in fractionating the venom and then applying an abstract name to the component. The venom also contains phospholipase, protease, collagenase, hyaluronidase, desoxyribonuclease, ribonuclease, and dipeptidases.

Bites by *Loxosceles* are very common in some parts of the world. For example, in Brazil, 3000 cases of *L. intermedia* envenomation are reported annually, and while more than 100 cases a year are attributed to *Loxosceles* in the United States, there is no doubt that other lesser known spiders or other arthropods or disease states are probably involved. Bites provoke pain in most cases, and a local burning sensation develops around the injury. Pruritus over the area often occurs, and the area becomes red with a small-blanching area surrounding the reddened bite site. The reddened area enlarges and becomes purplish during the subsequent 1 to 8 hours. It often becomes irregular in shape, and, as time passes, hemorrhages may develop throughout the area. A small bleb or vesicle may form at the bite site and increases in size. It can subsequently rupture, and a pustule form.

The whole area may become swollen and painful, and lymphadenopathy is common. If the central pustule ruptures, necrosis to various depths can be visualized. Laboratory work shows little change in the prothrombin and the partial thromboplastin times, while fibrinogen and platelets decrease early on, both subsequently returning to normal or in excess of normal. The hypofibrinogenemia, thrombocytopenia, and increased fibrinogen–fibrin degradation are thought to be a consequence of disseminated intravascular coagulation.

Other spiders

These have been noted elsewhere. The most important of these are *Steatoda*, *Chiracanthium*, *Phidippus*, and, perhaps, *Olios*. A word of caution gleaned from more than 25 litigations in which a spider was thought to be involved: If no spider is displayed or identified by an entomologist or an arachnologist in court, the evidence for “spider envenomation” is very much circumstantial.

Ticks

The question as to the “venomousness” of ticks, aside from the trauma of their bite, the possibility of secondary infection, the formation of a granuloma around the bite saliva, lymphocytoma, vectorship of microorganisms (Rocky Mountain Spotted Fever, tularemia, etc.), and other complications of tick bites, has been raised. The bite of some ticks, particularly *Ixodes holocyclus*, is known to contain a toxin and to produce flaccid paralysis and even death. Tick paralysis is known in both domestic animals and humans and has been noted in humans since 1912; however, tick envenomation was familiar to the American Indians (“Pajaroella,” *Ornithodoros corisceus*) long before that time. In most cases, it is an *Ixodes* sp. that is involved; however, in humans, the paralysis has been attributed to *Rhipicephalus simus*, *Hyalomma truncatum*, *Ixodes rubicundus*, and *Haemaphysalis*. In North America, the principal culprit in animals has been *Dermacentor andersoni*, while *Dermacentor variabilis* has been responsible for human envenomations in the Atlantic seaboard states. In most cases, it is the female tick that is involved. Ticks are basically of two types: the Iodidae or hard ticks, and the Argasidae or soft ticks. They are persistent, slow-feeding bloodsuckers that attach themselves firmly to their host. In general, they feed on warm-blooded animals including humans. The exact chemical nature of the toxin has not been elucidated, but it is thought to be a temperature-dependent toxin that inhibits evoked acetylcholine release at the neuromuscular junction.

Bites are often not felt, and the first evidence of envenomation may not be for several days when small

macules develop. The macules are 3 to 4 mm in diameter, surrounded by erythema, swelling, and often display a hyperemic halo. The patient often complains of difficulty with gait, followed by paresis, and, sometimes, eventually paralysis. Problems in speech and respiration may ensue and lead to respiratory paralysis if the tick is not removed. Because the tick is often in the hair, it may remain unseen and the differential diagnosis confused. Removal of the tick usually results in a rapid and complete recovery although regression of paralysis may resolve slowly.

It seems probable that the ticks that cause the paralysis in humans and domestic animals may be the same and that it is the length of the exposure to the feeding tick that determines the degree of poisoning. Obviously, the first signs of poisoning are less likely to be observed in cattle, sheep, dogs, and cats than in humans, and, as for symptoms (what the patient tells you), that is not likely to occur. Treatment consists of removal of the tick, using a formamidine derivative or petroleum product, washing with soap and water, and treating specifically for the paralysis or other manifestation. It should be pointed out, again, that these comments are only specific for tick venom poisoning and not for allergic reactions, transmission of disease states, or other tick bite complications.

CHILOPODA. CENTIPEDES

These elongated, many-segmented brownish-yellow arthropods are found worldwide (Fig. 2). They have a pair of walking legs on most segments, are fast moving, secretive, and nocturnal. They feed on other arthropods and even small vertebrates and birds; they are cannibalistic. The first pair of legs behind the head is modified into poison jaws or maxillipeds. Centipedes range in length from 3 to almost 300 mm. In the United States, the prevalent biting genus is a *Scolopendra* sp. The venom is concentrated within the intracellular granules, discharged into vacuoles of the cytoplasm of the secretory cells, and then by ducts that carry the venom to the jaws.

The venom of the centipedes contain high molecular weight proteins, proteinases and esterases, 5-hydroxytryptamine, histamine, lipids, and polysaccharides. In humans, some species produce cardiovascular

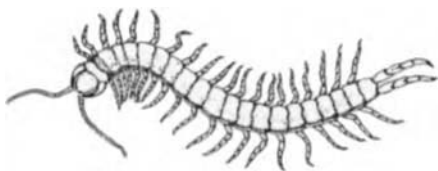


Fig. 2 CHILOPODA, Centipedes.

changes and alterations associated with acetylcholine release. The venom produces bleeding, redness, and swelling often lasting 24 hours. Localized tissue changes and necrosis have been reported; in severe envenomations, nausea and vomiting, changes in heart rate, vertigo, and headache have all been noted. Treatment is nonspecific, but washing and the application of a cream containing hydrocortisone, diphenhydramine, and tetracaine (Itch Balm Plus, Sawyer) are of value.

DIPLOPODA. MILLIPEDES

These arthropods are cylindrical, wormlike creatures, mahogany to dark-brown or black in color, bearing two pairs of jointed legs per segment, and ranging in length from 20 to 300 mm (Fig. 3). In some parts of the world, particularly Australia and New Guinea, the repugnatorial secretions expelled from the sides of their bodies contain a toxin of quinone derivatives and a variety of complex substances, such as iodine and hydrocyanic acid, which the animal makes use of to produce hydrogen cyanide. Some species can spray these defensive secretions, and eye injuries are not uncommon but rare in the United States.

The lesions produce by millipedes are generally known as “burn” injuries and consist of a burning or prickling sensation and the development of a yellowish or a brown-purple lesion with the formation of a subsequent blister containing serosanguinous fluid. They may rupture. Contact with the eye can cause acute conjunctivitis, periorbital edema, keratosis, and much pain; it must be treated immediately. Skin treatment consists of washing, washing, and washing the area thoroughly with soap and water and applying the cream as previously mentioned.

INSECTA

Lepidoptera. Caterpillars and Moths

The urticating hairs, setae, of caterpillars are effective defensive weapons that protect some species from predators (Fig. 4). The setae are attached to unicellular poison glands at the base of the hair. Both the larvae and the adult are capable of stinging, either by direct



Fig. 3. DIPLOPODA, Millipedes.



Fig. 4 INSECTA: Butterflies, Moths, Caterpillars.

contact with the setae or indirectly by the airborne irritation from the hairs. It appears that contraction of the abdominal muscles is sufficient to release the barbs from their sockets so that they can become airborne. Some caterpillars have a disagreeable smell or taste and are avoided by birds and other animals. The toxin found in the venom glands of some caterpillars may be derived from their feeding on noxious plants and then metabolized. Earlier studies showed the toxic materials contained aristolochic acid, cardenolides, and histamine, among other substances. In recent studies, fibrinolytic activity has been found at 16 and 18 kDa. Coagulation defects, such as prolonged prothrombin and partial thromboplastin times and a decrease in fibrinogen and plasminogen, have been noted.

In some parts of the world, the stings of several species of Lepidoptera give rise to bleeding, often severe and sometimes fatal. In the United States, envenomation by members of the family Saturniidae, the buck moths, the grapeleaf skeletonizer (family Zygaenidae), the puss moth (family Megalopygidae), and the brown-tailed moth *Euproctis* sp. generally give rise to a little more than immediate localized itching and pain usually described as burning, followed, in some cases, by urticaria, edema, and, occasionally, fever. In the more severe cases abroad, often due to *Megalopygie*, *Dirphia*, *Automeris*, and *Hemileuca* spp., there is localized pain, papules (sometimes hemorrhagic), hematomas, and, on occasions, headache, nausea, vomiting, hematuria, lymphadenitis, and lymphadenopathy. Cerebral edema hemorrhage (intracranial hypertension) and mental changes have been noted for foreign species.

Treatment consists of pressing cellophane tape to the affected area, removing it, doing it again, washing the area with warm soap and water, repeating this, and then applying the cream previously mentioned. Serious stings need to be treated specifically by a physician.

Hymenoptera. Ants, Bees, Wasps, Hornets, and Yellow Jackets

The economic, agricultural, and social problems of the Hymenoptera have been noted elsewhere in this text. The number of stings by ants per year circumglobally

must number in the tens of millions, while the stings of bees, wasps, yellow jackets and hornets must certainly number in the millions. In the United States, these animals are responsible for more deaths than all the other venomous animals. Because of the medical and the chemical importance of Hymenoptera and their venoms, they have received far more attention than the other venomous arthropods. Venoms of the ants, bees, and social wasps are used by these insects to defend themselves, while the solitary wasps generally use their venom to paralyze, not to directly kill their prey. Because of the diversity in venoms of the different Hymenoptera, only a brief summary of the biology, the chemistry, the pharmacology, and the clinical problem of their importance can be presented.

Formicidae. Ants

The stinging properties of the ants need no introduction (Fig. 5). Most species sink their powerful mandibles into the flesh providing leverage, then drive their sting into the victim. Most ants have stings, but those that lack a sting can spray a defensive secretion from the tip of their gaster which is often placed in the wound of the bite. Ants of the different species vary considerably in length, ranging from less than 1.5 to over 35 mm. In the United States, the clinically important stinging ants are the harvesting ants, *Pogonomyrmex*; the fire ants, *Solenopsis*; and the little fire ants, *Ochetomyrmex*. The harvester ants are large, red, dark-brown, or black, ranging in size from 6 to 10 mm, and having fringes of long hair on the center of their heads. They are vicious stingers, and their venom is said to have strong cholinergic properties.

The venoms of the ants vary considerably. The venoms of Ponorinae and Ecitoninae, as well as Pseudomyrmex, are proteinaceous in character. The Myrmecinae venoms are a mixture of amines, enzymes, and proteinaceous materials, histamine, hyaluronidase, and phospholipase A. Formicidae ant venom contains about 60% formic acid. Fire ants are unique in that while their venom is poor in polypeptides and proteins, it is rich in alkaloids which appear to be responsible for the pruritic pustules and necrosis. The sting of the fire ant gives rise to a painful burning sensation to which a wheal and localized erythema develop, leading in a few hours to a clear vesicle. Within 12 to 24 hours, the fluid becomes purulent, and the lesion becomes a pustule.

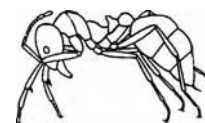


Fig. 5 Formicidae, Ants.

It may break down or become a crust of fibrotic nodule. In multiple stings, there may be nausea, vomiting, vertigo, increased perspiration, respiratory difficulties, cyanosis, coma, and even death. Treatment of ant stings is dependent upon their numbers, whether an allergic reaction is involved, or whether there are complications.

Apidae. Bees

In this family, we include the bumble bees, honey bees, carpenter bees, wasps, hornets, and yellow jackets (Fig. 6). The commonest stinging bee is *Apis mellifera*, but with the introduction and rapid spread of the Africanized bee, *Apis mellifer scutellata*, in the United States, the incidence of Hymenoptera poisonings is increasing. In 1996, there were at least 58 deaths and more than 1000 incidences of Africanized bee stings in Mexico and the United States. The venom of Africanized bee is different from that of the European bee, *A. mellifer mellifer*. The former bee is smaller and gives less venom, but its aggressiveness is such that attacks of 50–500 bees are not unusual. The overwhelming dose of apamine, which is thought to be a lethal factor, results in the serious or even fatal poisoning by this arthropod. In addition to apamine, the venom contains biologically active melittin synergized by phospholipase A₂, hyaluronidase, histamine, dopamine, and a mass cell-degranulating peptide, among other components. It is said that 50 stings can be serious and lead to respiratory dysfunction, intravascular hemolysis, hypertension, myocardial damage, hepatic changes, shock, and renal failure. With 100 or more stings, death can occur. A novel Fab-based anti-venom for massive bee attacks has been reported, but has not undergone clinical trial at the time of this writing. It could be valued in cases where the patient survives the initial onslaught of the poisoning and before serious sequelae develop.

Hemiptera. True Bugs

The clinically most important of the true bugs are the Reduviidae (the reduviids), the kissing bug, the assassin bug, the wheel bug, or the conenose bug of the genus *Triatoma*. Generally, they are a parasite of rodents and are common in wood rat nests or wood

piles. They are elongated bugs with a freely moveable cone-shaped head and a straight beak. The most common true bugs that are involved in bites are *Triatoma protracta*, *Triatoma rubida*, *Triatoma magista*, *Reduvius personatus*, and *Arilus cristatus*. During their nocturnal dispersal flights, they are attracted to porch or artificial light. Indeed, at our ranch in Portal, AZ, we have captured more than 100 reduviids in a single night using bright artificial light. The average length of these bugs was 19 mm (Fig. 7).

The venom appears to have apyrase activity and lacks 5-nucleotidase, inorganic pyrophosphatase, phosphatase, and adenylate kinase activities, but is fairly rich in protease properties. It inhibits collagen-induced platelet aggregation. It is said to contain a protein of 16–19 kDa.

The bites of *Triatoma* sp. are definitely painful and give rise to erythema, pruritis, increased temperature in the bitten part, localized swelling, and, in those allergic to the saliva, systematic reactions, such as nausea and vomiting, and angioedema. With some bites, the wound area will slough, leaving a depression. Treatment consists of cleansing the area and applying the cream previously described.

The water bugs are water-dwelling true bugs of which at least three families—Naucoridae, Belostomatidae, and Notonectidae—are capable of biting. They are found in lakes, ponds, marshes, quiet freshwater, and swimming pools. The most common biter in the United States is *Lethocerus americanus*, a Belostomatidae, ranging in length from 12 to 70 mm although some water bugs may reach 150 mm. The dorsum is usually tan or brown although it may be brightly colored, while the venter is brown. They are very strong insects and can immobilize snails, tadpoles, salamanders, even small fish, and water snakes. They are sometimes known as “toe biter” or “electric light bug.”

The venomousness of the water bugs has been attributed to their saliva, which is said to contain digestive enzymes, neurotoxic components, and hemolytic fractions. ApoLp-III has been isolated from the hemolymph of *Lathocerus medius*. It has a M_r of 19,000 and an amino acid composition high in methionine. If molested, water bugs will bite, and some species can bite in or out of the water. Their bites give rise to immediate pain, some localized swelling, and, in



Fig. 6 Apidae, Bees.

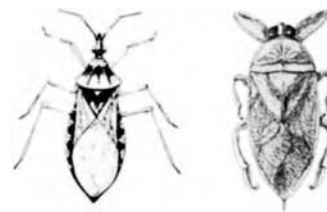


Fig. 7 Hemiptera, True bugs.

one case seen by the author, induration and the formation of a small papule. Treatment consists of cleansing the areas and applying the cream previously noted.

There are some arthropods that are poisonous as opposed to venomous; that is, they have no mechanism for delivering their toxin, the poison comes out when they're crushed or eaten. These would include, among others, the darkling or stink bugs (*Eleodes*), and the blister beetles (*Epicauta*) for which cantharidin is known.

BIBLIOGRAPHY

- Attygalk, A.B.; Morgan, E.D. Chemicals from the glands of ants. *Chem. Soc. Rev.* **1989**, *13*, 245–278.
- Bascur, L.; Yevenes, I.; Baria, P. Effects of *Loxosceles laeta* spider venom on blood coagulation. *Toxicon* **1982**, *20*, 795–796.
- Blum, M.S. Poisonous ants and their venoms. In *Insect Poisons, Allergens, and Other Invertebrate Venoms*; Tu, A., Ed.; Marcel Dekker: New York, 1984; 225–242.
- Buffkin, D.C.; Russell, F.E. A study of the venom of the imported fire ant: physiopharmacology, chemistry and therapeutics. *Proc. West. Pharmacol. Soc.* **1974**, *17*, 223–226.
- Cooper, B.J.; Spence, I. Temperature-dependent inhibition of evoked acetylcholine release in tick paralysis. *Nature* **1976**, *263*, 693–695.
- Deslippe, R.J.; Guo, Y.-J. Venom alkaloids of fire ants in relation to worker size and age. *Toxicon* **2000**, *38*, 223–232.
- Dos Santos, V.S.P.; Franco, C.R.C.; Viggiano, R.L.L. Structural and ultrastructural description of the venom gland of *Loxosceles intermedia* (brown spider). *Toxicon* **2000**, *38*, 265–285.
- Feitosa, L.; Gremski, W.; Veiga, S.S. Detection and characterization of metalloproteinases with gelatinolytic, fibrinolytic and fibrinogenolytic activities in brown spider (*Loxosceles intermedia*) venom. *Toxicon* **1998**, *36*, 1039–1051.
- Gertsch, W.J. *American Spiders*; Van Nostrand Reinhold: New York, 1979; 274 pp.
- Gibly, R.; Williams, M.; Walter, F.G.; McNally, J.; Conroy, C. Continuous intravenous infusion for *Centruroides exilicauda* scorpion envenomation. *Ann. Emerg. Med.* **1999**, *34* (5), 620–625.
- Harwood, R.F.; James, J.T. *Entomology in Human and Animal Health*, 7th Ed.; Macmillan: New York, 1979; 548 pp.
- Kaire, G.H. Isolation of tick paralysis toxin from *Ixodes holocyclus*. *Toxicon* **1966**, *4*, 91–97.
- Keegan, H.L. *Scorpions of Medical Importance*; University Press: Jackson, MS, 1980; 140 pp.
- Lockey, R.F. Systemic reactions to stinging ants. *J. Allergy Clin. Immunol.* **1974**, *54*, 146–152.
- Menez, A.; Zimmerman, K.; Zimmerman, S.; Heathwole, H. Venom apparatus and toxicity of the centipede *Ethmostigmus rubripes*. *J. Morphol.* **1990**, *206*, 303–312.
- Nutting, W.B. *Mammalian Diseases and Arachnids*; CRC Press: Boca Raton, FL, 1984; Vol. 2, 280 pp.
- Piek, T., Ed.; *Chemistry and Pharmacology of Honeybee Venom. Venoms of the Hymenoptera. Biochemical, Pharmacological and Behavioural Aspects*; Academic Press: Orlando, FL, 1986; 570 pp.
- Polis, G.A., Ed.; *The Biology of Scorpions*; Stanford University Press, 1990; 482 pp.
- Ribeiro, J.M.; Garcia, E.S. Platelet antiaggregating activity in the salivary secretion of the blood sucking bug, *Rhodnius prolixus*. *Experientia* **1981**, *37* (4), 384–386.
- Russell, F.E. The Toxic effects of animal toxins. In *Toxicology. The Basic Science of Poisons*, 5th Ed.; McGraw Hill: New York, 1996; 801–839.
- Russell, F.E. Venomous bites and stings. In *The Merck Manual*; Merck & Co.: Rathway, NJ, 1992; 2565–2576.
- Russell, F.E.; Marcus, P.; Streng, J.A. Black widow spider envenomation during pregnancy. Report of a case. *Toxicon* **1979**, *17*, 188–189.
- Russell, F.E.; Gertsch, W.J. Letter to the editor. *Toxicon* **1982**, *21* (3), 337–339.
- Smith, D.S.; Russell, F.E. Structure of the venom gland of the black widow spider *Latrodectus mactans*. A preliminary light and electron microscopy study. In *Animal Toxins*; Russell, F.E., Saunders, P.R., Eds.; Pergamon: Oxford, 1967; 1–15.
- Smith, R.L. *Venomous Animals of Arizona*; College of Agriculture: Tucson, AZ, 1997; 134 pp.
- Winston, M.L. *Killer Bees. The Africanized Honey Bee in the America*; Harvard University Press: Cambridge, MA, 1992.

Pollution of the Environment

Joseph D. Cornell

College of Environmental Science and Forestry, State University of New York,
Syracuse, New York, U.S.A.

INTRODUCTION

Pesticides are designed to kill pests.^[1] Unfortunately, when pesticides are released into the environment, they also can kill and cause harm to non-target species. The harmful effects of pesticides on non-target species have been well known since the 1960s when Rachel Carson wrote the classic book “Silent Spring.”^[2] In “Silent Spring,” Carson documented the effects of pesticides on nontarget species and described many of the ways in which pesticides reach humans and other species. Although a determined effort was made by the chemical and pesticide industries to discredit her work,^[3] Carson’s landmark book led to a greater awareness by the general public—in the United States and abroad—of the dangers of the use of many pesticides and the pollution that they cause. The environmental and social costs caused by this pollution was estimated to be about \$8123 million each year in the United States alone.^[4] In addition, pesticides account for about 3 million cases of acute poisoning each year worldwide.^[5]

PESTICIDES AS POLLUTANTS

There are five main factors that allow pesticides to become pollutants and cause such widespread and costly damage. First, many pesticides are toxic to a wide range of organisms. Second, only a very small percentage of the amount of pesticides that are applied reach their intended target organisms. Instead, the vast bulk of pesticides that are applied become environmental pollutants. Once they are released, pesticides may be transported to long distances by both wind and water. In addition to the potential for being transported over great distances, many pesticides remain toxic for long periods of time. Last, and perhaps most importantly, the use of pesticides is pervasive.

DIRECT AND INDIRECT EFFECTS OF PESTICIDE POLLUTION

Pesticides directly cause a great variety of lethal and toxic effects in non-target species. Exposure to

pesticides and pesticide residues has been implicated in the deaths and declines of insect, fish, bird, mammal, and amphibian populations worldwide. Pesticide poisoning is responsible for many fatalities and health problems in humans each year as well. Pesticide residues have also been implicated in a wide variety of indirect effects such as the increase of epizootic infections in wild animals, and ecosystem-level effects caused by the loss of predators, pollinators, and keystone species.

THE ECOLOGICAL EFFECTS OF PESTICIDE POLLUTION

One of the most fundamental ways in which pesticides affect the ecology of living organisms is through “bioaccumulation,” which occurs when pesticide residues accumulate in pest and non-pest species. Many pesticides are fat-soluble and accumulate in fatty tissues. Over time, as organisms feed on foods contaminated with pesticide residues, concentrations of these residues can reach toxic levels. The process of bioaccumulation can continue at each level of the food chain in an ecosystem, resulting in “biomagnification.” As predators at one trophic level feed on organisms at the next lower level, pesticide residues are concentrated, magnifying the effects of bioaccumulation at successive trophic levels.

As a result of biomagnification, the concentrations of pesticide residues in top predators such as hunting and fishing birds can reach levels that are hundreds, or even thousands, of times higher than the background levels and can cause the collapse of predator populations. For example, beginning in 1949, DDD was applied at a concentration of 0.02 ppm to control the gnat, *Chaoborus astictopus*, in Clear Lake, California. The pesticide was biomagnified within the ecosystem until by 1954, when levels of DDD in the fat of the top predator, the Western Grebe, had reached 1600 ppm. This resulted in the local extermination of the entire grebe population.^[6]

The loss of predators as a result of pesticide poisoning can also actually lead to the proliferation of the pests that the pesticides were originally meant to control. In many ecosystems, “natural enemies,” such as

predators, parasites, and competitors, help maintain the dynamic equilibria between pest and non-pest species. However, most pest species are classic r-strategists^[7] and are capable of reproducing much more quickly than their predators. When the application of pesticides does more harm to a pest's natural enemies than it does to the pest, populations of those pests can grow unchecked, leading to the resurgence of the pest species.

The removal of the natural enemies can also lead to the emergence of "secondary pest" species. For example, the brown planthopper (*Nilaparvata lugens*), originally a minor pest of rice, cost Indonesia over \$1 billion in rice losses during the 1970s after pesticides had eliminated the brown planthopper's predators and competitors.^[8]

The loss of predators because of pesticides can also lead to changes in the structure of the entire ecosystems by removing the "keystone species." Predators in many ecosystems exert a "top-down" control on other organisms. Because of biomagnification, predators are often at particular risk from pesticide pollution, as are the ecosystems that they inhabit. Pesticides have also been implicated in the long-term decline of another type of important "keystone" species: Pollinators. Both industrial agriculture and nature depend on the insects and other organisms that pollinate crops and wild plants alike. Unfortunately, populations of pollinators are declining in many countries engaged in industrialized agriculture, in part because of disease and in part because of pesticide poisoning. In the United States, bee populations are particularly important pollinators and are very susceptible to many widely used pesticides such as nealed.^[9] In 1981, Flint and van den Bosch^[6] estimated that in California, pesticides accounted for half of the decline in bee populations.

THE EFFECTS ON WILDLIFE

Ever since the publication of "Silent Spring," perhaps the clearest threat posed by pesticide pollution, especially in the minds of the general public, is the threat to wildlife. For example, exposure to pesticides in general, and DDT in particular, was blamed for the collapse of predatory bird population in the 1950s and 1960s throughout Europe and North America.^[10] Although the use of DDT was banned in the United States in 1972 and was banned in most western European countries in the 1970s, some populations of predatory birds still have not recovered from these losses.

Likewise, exposure to pesticides has been invoked to explain the recent global occurrence of amphibian deformities,^[11] the decline in amphibian populations worldwide, and the extinction of some amphibian species.^[12] For example, in the Sierra Nevada Mountains

of California, pesticide residues from the valley below have been detected in precipitation as high as 2200 m and have been linked to the local loss of amphibian populations. Most amphibians must complete at least part of their life cycle in water, which if contaminated, insures exposure to pesticide residues. In addition, amphibians have thin, permeable skins, which readily absorb organic compounds, as do their eggs, which lack protective shells. Their ecological, phenological, and morphological characteristics therefore would seem to make amphibians particularly susceptible to the effects of pesticide pollution.

Marine mammals are another group of animals that seem to be particularly at risk because of exposure to pesticides. Marine environments are subject to both local sources of pollution and the long-range transport of pollutants. Marine mammals are often top predators that are subject to the effects of bioaccumulation and biomagnification. Moreover, marine mammals have higher percentages of body fat relative to other mammal species. The percentage of body fat is important because many organic pesticides are fat-soluble and accumulate in fatty tissue. In addition, it was shown that fat-soluble pesticides are passed from mother to offspring in some groups of marine mammals such as baleen whales.^[13] All these factors have led a growing number of researchers to suggest that pesticide residues are contributing to an upsurge in mortality in marine mammals,^[14] including deaths by infectious disease. In Russia, high levels of DDT were found in the blubber of thousands of Caspian seals, which died from canine distemper virus.^[15] Some researchers have suggested that the DDT, by compromising the seal's immune system, may be linked to the deaths. However, no definitive studies as yet have conclusively demonstrated this linkage.

THE FUTURE POTENTIAL FOR PESTICIDE POLLUTION

The incidence and severity of environmental pollution as a result of the use of pesticides has the potential to enormously grow in the near future. There are many reasons for the potential growth of environmental pollution from pesticides, but the most important reason will be the continued growth of human populations, which will create incredible pressures on agricultural production systems to keep up with the demand for food. Increased demand for food will almost certainly be met by attempts to increase both the extent and intensity of agriculture worldwide.^[16] Pesticides have become so integrated into agricultural production that many farmers do not believe that they have any economically feasible alternative to their use.^[17] Therefore, increasing the areal extent of food production will

likely expose increasingly larger areas to environmental pollution from pesticides, especially as we encounter limits to alternative pest control methods such as biocontrol.

Increasing the extent of industrialized agriculture will also reduce the “patchiness” of the agricultural landscape, conceivably making it more vulnerable to invasion by weeds, insects, and pathogens, which in turn will most likely be met by increases in the application of pesticides. Likewise, increasing the intensity of global agriculture will almost certainly include increases in the rate of application of all kinds of pesticides. As the total amount of pesticides being applied increases, the potential for environmental pollution will increase as well.

In addition, other related factors will almost certainly lead to massive increases in the global use of pesticides in the future. One example is the expected response to the emergence and spread of infectious disease.^[18] Increased risks from infectious disease come with increases in globalization and rapid transportation.^[19] Many of the most disturbing of these diseases are spread by insects. Therefore, efforts to safeguard public health have heavily relied on the use of pesticides to control the spread of disease vectors. Pesticides have been used to control outbreaks of St. Louis encephalitis,^[20] eastern equine encephalitis,^[21,22] and most recently, the West Nile virus, which spread from Africa to the northeastern United States in 1999.^[23]

Adding to the problem of controlling infectious diseases are the potential effects of climate change, which has the potential to alter the distribution of some insect-borne diseases such as malaria.^[19] Rogers and Randolph^[24] predict that by the year 2050, the climate of large portions of the southeast of North America, including the states of Florida, Louisiana, and Texas, will become more suitable for the spread of *Plasmodium falciparum*, the infectious agent that causes malaria, and the anopheline mosquitoes that spread the disease. Again, one predictable response to this threat will be increased spraying of pesticides, although this may also increase environmental pollution and unwanted effects on humans and non-target species. Climate change also has the potential to alter the distribution of agriculture as a whole in North America and throughout the rest of the world and drastically shift the distribution of pest species.

BIOINVASIONS

Pesticides will also almost certainly be increasingly used to control the so-called bioinvasions of exotic plants, animals, and crop pathogens. Again, as human populations have grown and become more mobile, and as international commerce expands, exotic organisms

such as the zebra mussel and the Asian longhorned beetle have spread into new areas.^[25] Efforts to control these pests have been largely limited to conventional pesticides despite the unintended, but by now well-known, effects of these pesticides on the environment and human health. For example, efforts to control the Mediterranean fruit fly (or “medfly”), introduced into this country in the 1980s, have heavily relied on aerial spraying of pesticides such as Malathion. In 1999, in Florida alone, there were 123 cases of respiratory, gastrointestinal, neurological effects, dermatitis, and eye damage linked to the spraying of Malathion for medfly control.^[26]

CONCLUSION

As long as humans are dependent on industrialized agriculture, we will need to continue to use traditional chemical pesticides. Therefore as long as chemical pesticides remain toxic to non-target species, pesticides and pesticide residues will continue to be serious pollutants even when applied using the best practices and under the best conditions. Because of the potential growth of pesticide use in the future, particularly in the developing world, it is increasingly unlikely that we will always be using these pesticides under the best of conditions or using the best possible practices. Instead, it is much more likely that in the future, we will see increases in both the total use of pesticides and increases in the pollution that they can cause. However, our understanding and awareness of the effects of pesticide pollution has grown substantially since Rachel Carson’s “Silent Spring.” It is possible that if we continue to recognize their potential hazards, we can also continue to mitigate their effects.

REFERENCES

1. According to the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA), a pesticide is “any substance or mixture of substances intended for preventing, destroying, repelling, or mitigating any pest” (<http://www.epa.gov/pesticides/fifra.htm>).
2. Carson, R. *Silent Spring*; Houghton Mifflin: New York, 1962.
3. Walker, M.J. The unquiet voice of ‘Silent Spring’: the legacy of Rachel Carson. *Ecologist* **1999**, *29* (5), 322–325.
4. Pimentel, D.; Acquay, H.; Biltonen, M.; Rice, P.; Silva, M.; Nelson, J.; Lipner, V.; Giordano, S.; Horowitz, A.; D’Amore, M. Environmental and economic costs of pesticide use. *BioScience* **1992**, *42* (10), 750–760.
5. Jeyaratnam, J. Acute pesticide poisoning: a major global health problem. *World Health Organization (WHO) Stat. Q.* **1990**, *43*, 139–144.

6. Flint, M.L.; van den Bosch, R. *Introduction to Integrated Pest Management*; Plenum Press: New York, 1981.
7. Conway, G. Man versus pests. In *Theoretical Ecology*; May, R.M., Ed.; Blackwell Scientific Publications: Oxford, 1981; 356–386.
8. Gardner, G. IPM and the war on pests. *WorldWatch* **1996**, *9* (2), 22–27.
9. <http://www.epa.gov/pesticides/citizens/naledfs.pdf>
10. Lensink, R. Range expansion of raptors in Britain and the Netherlands since the 1960s: testing an individual-based diffusion model. *J. Anim. Ecol.* **1997**, *66*, 811–826.
11. Ouellet, M.; Rodrigue, J.; Bonin, J.; Lair, S.; Green, D.M. *Developmental Abnormalities in Free-Living Anurans from Agricultural Habitats*; Canadian Wildlife Service: Québec Region, October, 1998. http://www.qc.ec.gc.ca/faune/faune/html/malformations_e.html.
12. Mattoon, A. Amphibia fading. *WorldWatch* **2000**, *13* (4), 12–23.
13. Aguilar, A.; Borrell, A. Reproductive transfer and variation of body load of organochlorine pollutants with age in fin whales (*Balaenoptera physalus*). *Arch. Environ. Contam. Toxicol.* **1994**, *27* (4), 546–554.
14. Colborn, T.; Smolen, M. Epidemiological analysis of persistent organochlorine contaminants in cetaceans. *Rev. Environ. Contam. Toxicol.* **1996**, *146*, 91–172.
15. Stone, R. Canine virus blamed in Caspian seal deaths. *Science* **2000**, *289*, 2017–2018.
16. Tinker, P.B. The environmental implications of intensified land use in developing countries. *Philos. Trans.-R. Soc., Biol. Sci.* **1997**, *352*, 1023–1033.
17. Weber, P. A place for pesticides? *WorldWatch* **1992**, *5* (3), 18–25.
18. Pimentel, D.; Tort, M.; D'Anna, L.; Krawic, A.; Berger, J.; Rossman, J.; Mugo, F.; Doon, N.; Shriberg, M.; Howard, E.; Lee, S.; Talbot, J. Ecology of increasing disease. *BioScience* **1998**, *48* (10), 8-17-826.
19. Platt, A. The resurgence of infectious disease. *World-Watch* **1995**, *8* (4), 26–32.
20. Kilpatrick, J.W.; Adams, C.T. Emergency Measures Employed in the Control of St. Louis Encephalitis Epidemics in Dallas and Corpus Christi, Texas, Proceedings 23rd Annual Meeting American Mosquito Control Association, 1966; 1967.
21. Edman, J.D.; Timperi, R.; Werner, B. Epidemiology of eastern equine encephalitis in Massachusetts. *J. Fla. Mosq. Control Assoc.* **1993**, *64*, 84–96.
22. Howard, J.J.; Morris, C.D.; Emord, D.E.; Grayson, M.A. Epizootiology of eastern equine encephalitis in upstate New York, USA. VII. Virus surveillance 1978–85, description of 1983 outbreak, and series conclusions. *J. Med. Entomol.* **1988**, *25*, 501–514.
23. U.S. Centers for Disease Control. **2000**. <http://www.cdc.gov/ncidod/dvbid/westnile/index.htm>.
24. Rogers, D.J.; Randolph, S.E. The global spread of malaria in a future, warmer world. *Science* **2000**, *289*, 1763–1766.
25. Bright, C. Bioinvasions: the spread of exotic species. *WorldWatch* **1995**, *8* (4), 10–19.
26. U.S. Centers for Disease Control. Surveillance for acute pesticide-related illness during the Medfly eradication program, Florida, 1998. *Morb. Mort. Wkly. Rep.* **1999**, *48* (44), 1015–1027.

Prescriptive Use of Pesticides

Harold Coble

Department of Crop Science, North Carolina State University, Raleigh, North Carolina, U.S.A.

INTRODUCTION

The medical profession operates under a model whereby relatively low-risk pharmaceutical chemicals may be purchased and used without prescription, but high-risk chemicals may be used only after prescription by a trained and licensed professional (physician). A similar prescriptive use scenario may be a mechanism by which certain valuable but high-risk pesticide uses could be maintained as an alternative to cancellation of use of those products. Comparing pesticides to human medications and the accompanying prescriptive practices is a useful exercise, although there are some very basic differences in operational applications. A pharmaceutical product typically evolves from a tightly controlled prescription to a general prescription, and potentially to over-the-counter use. Conceptually, all registered pesticides currently are for over-the-counter use, although restricted-use products require dispensing by a licensed dealer. Pharmacists are licensed dealers required to meet national standards and are licensed through state testing, licensing, and enforcement programs. This process is comparable to the manner in which pesticide dealers and consultants are licensed with respect to general-use and restricted-use compounds. However, in agriculture there is presently no equivalent to the physician, who can provide recommendations for improved human health through the ability to draw from a broad list of compounds, some of which are controlled expressly because they have potentially serious side effects if not properly managed.

PRESCRIPTION USE CLASSIFICATION OF PESTICIDES

In both environmental and human health risks, control of exposure to toxicants will result in control of risk. Monitoring volume of use, through prescriptions for compounds under risk pressure, could offer a means of assuring the EPA that an appropriate margin of safety relative to such compounds is maintained over time. Most states already have pesticide use record-keeping requirements but few have reporting requirements. Changes would be needed in reporting requirements to give the EPA a mechanism to monitor

and control exposure. Enhancing existing state programs through voluntary efforts recognized and supported by the EPA and industry may be one way to provide both the protection crops need and the protective mechanism that the EPA needs for certain compounds. Developing such a program would require the definition of qualifications for individuals who could administer those products.

QUALIFICATIONS OF PRESCRIBERS

Currently, each state has requirements for pesticide certification. The programs conform to national standards but include state-developed testing schemes, training programs, and licensing arrangements. Generally, certification depends on the ability of an individual to pass a specific qualifying exam at various criteria levels (applicator, operator, consultant). Recertification and continued licensing depends on documented attendance at qualifying training sessions for a given amount of time. Several options exist for a simple mechanism to provide qualified individuals capable of issuing pesticide use prescriptions. One option could be minimum standards for education or experience requirements. Another option could be an increased requirement for continuing education or a more rigorous exam that includes test criteria on compounds identified as prescriptive. A more rigorous exam or even a specific exam for qualification in prescription dispensing would allow those currently recommending curative or preventive measures, through the recommendations of a crop consulting service, to maintain and enhance their practice. Educational or experiential requirements may be more difficult to establish and could have the potential to disqualify otherwise qualified practitioners.

As far as the individuals qualifying as a prescriber, there is much debate. Manufacturer representatives generally have a wealth of product knowledge, but may be perceived as having a conflict of interest under a prescription scenario. USDA or state officials may be a source for prescribers, although state and federal resources to act in this capacity may be limited. Independent crop consultants may be the most logical source of prescribers, but their numbers are relatively

small compared to what would be needed under prescriptive use.

Just as medicine has moved from general practice to specialization, implementation of a prescription process could impact agricultural practitioners in the same manner. Given the relatively small pool of talent available in the agricultural plant health area, forcing specialization could be a detrimental limitation unless sufficient time is allotted to train the work force. Also, some degree of specialization naturally occurs because crop management is a local issue depending on climate, crop, pest pressures, and location of the growing area being managed. Recognition of the importance of localized experience is a critical factor to consider in a pesticide prescription process.

POTENTIAL IMPACTS

Farmers obviously would be the primary group impacted by prescriptive use of pesticides. Prescriptive use may be a way of assuring continued availability of certain pesticides important in the production of some commodities. If a pesticide, or certain uses of a pesticide, were found to exceed acceptable risk standards, one option under the Food Quality Protection Act of 1996 is for the EPA to cancel registration of that pesticide or use. If use of such a pesticide only by prescription were considered to fall within acceptable risk parameters, then the availability of that pesticide use could be continued. However, if prescriptions for pesticide use are required, there are at least three issues of major concern to farmers. First, there will be a cost associated with prescriptive use, and it is not clear who would bear that cost. Second, many pesticide use decisions must be made in a very short time after discovery of a pest infestation to avoid unacceptable

crop loss. Any delays caused by the necessity for prescriptions may be unacceptable in the context of good pest management practice. Third, introducing another aspect to use could expose the farmer to increased liabilities.

Agribusiness, including pesticide manufacturers, the distribution network, and dealers and suppliers would be impacted by prescriptive use. In addition to the challenge, based on need, of getting appropriate amounts of certain pesticides to the right place at the right time, manufacturers would have to decide the economic feasibility of supporting registrations for prescriptive use. Certainly, no manufacturer could afford to support a pesticide registration based solely on prescriptive use. However, most companies probably would support prescriptive use on their labels if sufficient nonprescriptive uses could be maintained to support profitable production and distribution of the product. Any new or additional registrations for prescriptive use of a product probably would have to be handled through the Interregional Research Project No. 4 (IR-4) because a manufacturer probably could not justify the cost associated with obtaining new registrations for such limited potential use.

BIBLIOGRAPHY

- Goodell, P.B.; Zalom, F.G. Delivering IPM: progress and challenges. In *Emerging Technologies for Integrated Pest Management*; Kennedy, G.G., Sutton, T.B., Eds.; APS Press: St. Paul, MN, 1999; 483–496.
- Kennedy, D. Humans in the chemical decision chain: politics and risk. In *Chemicals in the Human Food Chain: Sources, Options, and Public Policy*; Carter, H.O., Nuckton, C.F., Eds.; Agricultural Issues Center, University of California: Davis, 1988; 9–19.

Protected Crops Pest Management: Insects and Mites

Ramon Albajes

Department of Entomology, Centre UdL-IRTA, University of Lleida, Lleida, Catalonia, Spain

INTRODUCTION

Protected crops are affected worldwide by a number of insect and mite pests, mostly whiteflies, aphids, dipteran leafminers, caterpillars, and spiders, eriophyid, and tarsonemid mites (Table 1). Chemical control is still the prevalent method of pest control in plastic houses and tunnels. However, significant advances in implementing integrated pest management (IPM) systems—based on biological and microbial control, cultural practices, and selective use of pesticides—have been made in recent years. A wider use and integration of other IPM tools, particularly host-plant resistance, may be expected in the near future. The development of methods more adapted to local conditions and requirements as well as the organization of research, extension, and advisory networks are considered as key aspects to be improved in plastic-house growing areas for a faster implementation of integrated pest management programs.

PROTECTED CROPS WORLDWIDE

A variety of structures and types of covers are used to protect vegetable and ornamental crops against adverse—mostly climatic—conditions.^[1] Both low and high tunnels covered by plastic films and plastic houses are common structures of protected crops in warm regions like the Mediterranean Basin (185,000 ha) and eastern and southeastern Asia (381,000 ha), whereas glasshouses are prevalent in colder areas like northern Europe (26,000 ha). Complementary entries to which the reader is referred are (†) *Glasshouse Crop Pest Management* and (†) *Ornamental Crops*. Compared with glasshouses, plastic-covered houses and tunnels have several differential characteristics, including less climatic regulation, shorter and lower-yielding crop cycles (but frequently with two crops per year), and open structures that allow seasonal and even daily inside–outside exchange of pests and natural enemies.

The protected crop environment tends to favor plant growth and yield but may also enhance the rate of increase of herbivorous pests due to favorable climatic conditions, lack or delay in the establishment of natural enemies, and fast plant growth leading to foliage softness. Additionally, the increasing

international trade of ornamental plants leads the catalogue of new pests in protected crops to grow incessantly. Several years ago, most pests were easily controlled by chemical insecticides and miticides, but in recent years, a variety of factors are leading growers to adopt more integrated pest management systems.

MAJOR INSECT AND MITE PESTS AND CONTROL TACTICS

A number of insects and mites cause economic damage in protected vegetable and ornamental crops if no control measures are adopted. Although most pests are polyphagous, they commonly show preferences for certain crops and crop cultivars. Table 1 shows the most common insect and mite pests of Mediterranean protected crops, the most affected vegetable crops, and control tactics used within IPM systems.^[2–4] A few additional oligophagous pests may also damage specific crops. Biological and microbial controls, selective use of chemicals, and both greenhouse and crop management are the cornerstones of IPM in protected vegetable crops. Ornamental crops are more dependent on chemicals as economic thresholds are commonly near zero.

Biological and Microbial Controls

Insect and mite pest management in Mediterranean protected crops is still based on chemicals. However, significant progress in implementing more integrated technology has been made in several areas, usually around research institutes. Initial steps in the development of IPM systems attempted to adapt methods based on seasonal inoculative biological control that had been very successful in glasshouses in northern Europe.^[5,6] The substitution of chemicals by periodic releases of *Encarsia formosa* and *Phytoseiulus persimilis*—first in France and later in Spain and Italy—allowed the indigenous natural enemies to colonize protected crops and to show the potential of natural control in Mediterranean greenhouses. Whiteflies, leafminers, leaf-eating caterpillars, and spider mites are acceptably managed by natural, biological, or microbial controls, whereas successes in the biological control of aphids and thrips are still incipient. The development of biological control of secondary pests,

Table 1 Main insect and mite pests of protected vegetable crops. Major crops affected and natural enemies that can be managed or released

| Pest | Most affected crops | Control tactics and natural enemies that can be managed or released |
|---|-----------------------|--|
| Whiteflies | | BC, CGM, SP |
| <i>Trialeurodes vaporariorum</i> | Ab, Cb, Fb, Tm | <i>Encarsia formosa</i> , <i>Macrolophus caliginosus</i> , ^a <i>Dicyphus tamaninii</i> ^{ab} |
| <i>Bemisia tabaci</i> and <i>B. argentifolii</i> | Ab, Cb, Fb, Lt, Tm | <i>Eretmocerus eremicus</i> , ^b <i>Er. mundus</i> ^{ab} |
| Aphids, mainly | | SP, CGM, BC For all species: <i>Chrysoperla carnea</i> , <i>Aphidoletes aphidimyza</i> ^a |
| <i>Myzus persicae</i> | Most | <i>Aphidius matricariae</i> , ^a <i>A. ervi</i> |
| <i>Aphis gossypii</i> | Cb, Fb, Pp, | <i>Aphidius colemani</i> |
| <i>Macrosiphum euphorbiae</i> | Tm | <i>Aphelinus abdominalis</i> ^a |
| Thrips | | BC, CGM |
| <i>Frankliniella occidentalis</i> | Most | <i>Orius</i> spp., ^a phytoseiids, <i>D. tamaninii</i> ^{ab} |
| <i>Thrips palmi</i> ^c | | |
| Leafminers | Cb, Fb, Lt, Tm | BC, CGM |
| <i>Liriomyza trifolii</i> | | For all species: |
| <i>L. bryoniae</i> | | <i>Diglyphus isaea</i> ^a , <i>Dacnusa sibirica</i> ^d |
| <i>L. huidobrensis</i> | | |
| <i>L. sativa</i> ^c | | |
| Leaf-eating caterpillars | Most | MC, BC |
| <i>Spodoptera littoralis</i> | | |
| <i>S. exigua</i> | | |
| <i>Chrysodeixis chalcites</i> | | <i>Trichogramma</i> sp. ^b |
| <i>Autographa gamma</i> | | |
| Cutworms | Most | SP, CGM |
| <i>Agrotis</i> spp. | | |
| Spider mites | | BC, CGM |
| <i>Tetranychus urticae-cinnabarinus</i> | Ab, Cb, Fb, Sw | <i>P. persimilis</i> (Mediterranean strain ^a) |
| Eriophyoid mites | | CGM, SP |
| <i>Aculops lycopersici</i> | Tm | Phytoseiids ^{ab} |
| Tarsonemid mites | | CGM, SP |
| <i>Phytonemus fragariae</i> | Sw | <i>Neoseiulus cucumeris</i> ^a |
| <i>Polyphagotarsonemus latus</i> | Pp, Tm | Phytoseiids ^{ab} |

Keys for identifying crops: Ab = aubergine; Cb = cucurbits; Fb = French beans; Lt = lettuce; Pp = pepper; Sw = strawberry; Tm = tomato. Control tactics are mentioned by the following acronyms: BC = biological control; MC = microbial control; CGM = crop and greenhouse management; SP = selective use of pesticides.

^aNative of the Mediterranean area.

^bThe natural enemy is only applied at experimental scale.

^cInsect pests with risk of being introduced in the Mediterranean Basin.

^dOnly in areas under influence of Atlantic climate.

such as eriophyoid and tarsonemid mites, is at an earlier earlier stage.

Crop and Greenhouse Management

Crop and greenhouse management practices for pest control aim to modify the environment in order to

diminish the rate of increase of pest populations and enhance the activity of natural enemies. The cultural practices that should be considered in IPM in protected crops include soil plowing to interrupt the cycle of insects that have soil-inhabiting phases, like leafminers and thrips; management of greenhouse window openings to prevent the development of

diseases and to let in natural enemies; removing weeds in order to eliminate pest or virus reservoirs (but note that weeds may also be reservoirs of parasitoids and predators), avoiding overfertilization, which enhances fecundity of mites and some insects such as aphids, whiteflies, and leafminers; planning crop cycling to minimize pest migration or to take advantage of natural enemies that are already established; eliminating overluxurious foliage by pruning; and removing lower leaves only when parasitoids have already emerged. Greenhouse window screening to prevent pest entrance is practiced in some areas, but it may lead to greenhouse ventilation problems when screens are narrow-meshed as in the case of screens used for thrips and whitefly exclusion. Screens may additionally hinder the immigration of pest natural enemies onto the crop.

Selective Use of Chemicals

Most chemical interventions within IPM programs in protected crops are based on selective use of pesticides—early detection and treatment of first greenhouse colonization foci—when alternative methods are not available or not effective enough. This is the case of aphids or secondary pests like russet and broad mites. In recent years, the development of insect growth regulators (IGRs) has opened up new prospects for the use of insecticides in combination with biological and microbial control, particularly against whiteflies and caterpillars. However, as these products have become better known, they have been found to involve risks to natural enemies.

Other Tactics for IPM

Host-plant resistance has been largely unexploited as an insect management tool in protected vegetable and ornamental crops. Recent examples of the use of host-plant resistance to decrease rates of increase of pests or to make natural enemies perform better—as in the case of the tri-trophic cucumber–whitefly–*E. formosa* relationship—have shown the benefits of growing crop varieties specifically adapted to IPM programs. Host-plant resistance for plant virus control simplifies the biological control of virus vectors; this has been the case of tomato varieties resistant to TSWV, which have facilitated the biological control of *Frankliniella occidentalis*. Pheromones for mating disruption purposes in leaf-eating caterpillars are only under experimentation, although in the Mediterranean Basin, at least part of the lepidopteran populations probably mate outside the greenhouse.

FUTURE CONCERNS

Several stimuli are leading Mediterranean growers to adopt IPM systems:

- There is increasing consumer concern about chemical residues and unsustainable ways of producing food.
- Increasing pesticide-resistance problems are arising among the most damaging pests.
- As side-effects of pesticides become more evident, several initiatives to restrict their use are being undertaken by governments.
- Many nonchemical methods of controlling pests in protected crops have shown, when integrated, to be more effective than just spraying.
- The successful use of pollinators—mostly bumblebees—which are susceptible to most pesticides, is leading growers to refuse using chemicals that interfere with pollinator activity.

Biological control as practiced in glasshouses is largely inefficient and too expensive in Mediterranean plastic houses. Considering the greenhouse habitat as a component of a patched landscape with constant pest and natural enemy population, exchanges among protected plots, unprotected fields, and non-agricultural habitats (including field margins and woodlands) would probably be a more fruitful approach to identifying the k-factors that may be managed in order to reduce pest pressure on greenhouse crops; additionally, strategies to conserve and augment native parasitoids and predators in the area may favor natural control in greenhouses. Within such a framework, general predators that are able to successively colonize annual crops may be particularly useful. Banker-plant systems (in which potted plants, usually different to the crop, infested with alternative prey and the natural enemy are introduced in the greenhouse) could be implemented to establish natural enemies in the greenhouse early in the season or to maintain them in the greenhouse between successive cropping seasons. Unfortunately, natural control is sometimes unpredictable and thus unreliable. In this case, occasional releases of natural enemies that are adapted to local conditions and selective pesticide applications may be necessary. To avoid unnecessary releases of beneficials and chemical interventions, fast and reliable sampling and monitoring tools need to be developed. Locally adapted crop and greenhouse management practices should also help to keep pests at low densities. However, IPM will continue to be slowly adopted in the Mediterranean Basin if organizational aspects are not taken into account. Research, extension, and advisory systems are poorly developed in many Mediterranean countries, and in such situations, the mere transposition of exotic

schemes to local needs has proved to be dramatically inefficient.

REFERENCES

1. Wittwer, S.H.; Castilla, N. Protected cultivation of horticultural crops worldwide. *HortTechnology* **1995**, *5*, 6–23.
2. IOBC/WPRS Bulletin. Proceedings of Meetings of Working Groups on Integrated Control in Protected Crops. Issues since 1984: 8(1), 10(2), 12(3), 13(5), 14(5), 16(2), 16(8), 19(1), 17(5), 20(4), 22(1), 23(1).
3. Cavalloro, R., Pelarents, C., Eds.; *Integrated Pest Management in Protected Vegetable Crops*; Balkema: Rotterdam, The Netherlands, 1989.
4. Albajes, R., Gullino, M.L., van Lenteren, J.C., Elad, Y., Eds.; *Integrated Pest and Disease Management in Greenhouse Crops*; Kluwer Academic Publishers: Dordrecht, The Netherlands, 1999.
5. Hussey, N.W., Scopes, N.E.A., Eds.; *Biological Control. The Greenhouse Experience*; Blandford Press: Poole, Dorset, UK, 1985.
6. Heinz, K.M., van Driesche, R., Parrella, M.P., Eds.; *Biological Control of Arthropod Pests in Protected Culture*; Ball: Batavia, California, USA, 2002; *in press*.

Quality Control of Formulations

Árpád Ambrus

*Agriculture and Biotechnology Laboratory, International Atomic Energy Agency,
Vienna, Austria*

László Bura

Central Service for Plant Protection and Soil Conservation, Budapest, Hungary

INTRODUCTION

Pesticides have played an important role in the world's food production. They will remain indispensable in the foreseeable future in the economic production and post-harvest protection of sufficient amount of quality food for the continuously growing population of the world, and in the control of vectors of human and animal diseases.

The quality control of pesticide formulations is necessary to assure that the product is suitable for the intended use, and to avoid the undesirable agricultural, environmental, human-health, and social consequences of the use of inferior-quality products. It is a basic element of integrated crop protection and sustainable development, and should be carried out regularly.

WHAT IS A PESTICIDE FORMULATION?

The term formulation means the combination of various ingredients designed to render the product useful and effective for the purpose claimed. It contains the technical grade active ingredient(s) and the formulants. The active ingredient is the component of a formulation responsible for the biological activity against pests and diseases, or in regulating metabolism/growth, etc. A single active ingredient may be composed of one or more chemical or biological entities, which may differ in relative activity. A formulation may contain one or more active ingredients. A technical grade pesticide consists of the active ingredient as well as impurities which are by-products of the synthesis or derived from the raw materials used in the manufacturing process. Any substance, other than a technical grade active ingredient, intentionally incorporated in a formulation, is a formulant.^[1] The formulation chemistry and technology are described in a great number of books and publications. A good starting point could be one of the recent ones.^[2]

Pesticides are available in a wide range of formulation types.^[3] In addition to the classical formulations

such as dustable powder (DP), granules (GR), wettable powders (WP), emulsifiable concentrates (EC), a number of new formulations were developed which reduce occupational exposure and increase environmental safety. Some examples are encapsulated granule (CG), capsule suspension (CS), water-in-oil emulsion (EO), oil-in-water emulsion (EW), emulsifiable gel (GL), microemulsion (ME), water dispersible granule (WG).

QUALITY CONTROL OF PESTICIDES

The general quality requirements for pesticides are usually defined in national laws and regulations and specified for each product in the registration document or in the permit issued by the Government. The FAO and WHO pesticide specifications^[4] are designated to reflect generally acceptable quality criteria, against which products can be judged, either for regulatory purposes or in commercial dealings, or can be used where national registration does not specify quality parameters. They define the essential chemical and physical properties that may be linked to the efficacy and safe use of a product.

The quality control of the formulations includes, among others, the determination of the concentration of active ingredient(s), specified significant impurities, physicochemical parameters (Table 1), key coformulants, and storage stability. Generally, those impurities are tested which, compared with the active ingredient, are toxicologically significant to health or the environment, phytotoxic to treated plants, cause taint in food crops, affect the stability of the pesticide, or cause any other adverse effect.

Packaging and labeling are checked to ensure safe handling, storage without deterioration during the expected lifetime of the product, and provide sufficient information for its efficient and safe use, respectively.

National authorities are generally taking measures to ensure the compliance with quality specifications of pesticides. The official quality control aims to verify whether the formulations that have been placed on the

Table 1 Examples of physicochemical parameters

| Test | Aim of the test |
|---|---|
| Acidity, alkalinity or pH | To minimize potential decomposition of the active substance, deterioration of the physical properties of the formulation |
| Adhesion to seeds | To ensure that the intended dose remains on seeds and is not easily removed |
| Attrition resistance | To ensure that granular formulations remain intact until use |
| Degree of dissolution and/or solution stability | To ensure that water-soluble formulations dissolve readily and soluble concentrates produce stable solutions on dilution |
| Disintegration time | To ensure that soluble or dispersible tablets disintegrate rapidly on addition to water |
| Dispersion stability | To ensure that a sufficient amount of active substance is homogeneously dispersed in suspension and emulsion in the spray liquid |
| Dispersibility | To ensure that the formulation is easily and rapidly dispersed when diluted with water |
| Dissolution of water soluble bags | To ensure that formulations packed in water soluble bags, when dispersed or dissolved, will not block the filters or nozzles of application equipment |
| Dry sieve test | To restrict the content of particles of unwanted sizes |
| Dustiness | To restrict the dustiness of granular formulations, which may liberate dust into air when handled and applied |
| Emulsion stability | To ensure that a sufficient amount of active substance is homogeneously dispersed in emulsion |
| Flowability | To ensure that powders for direct application will flow freely from application machinery |
| Integrity | To ensure that tablets remain intact until use, to avoid risk from dust and to ensure that the intended dose is always applied |
| Nominal size range | To ensure that an acceptable proportion of a granule formulation is within an appropriate particle size range |
| Persistent foam | To limit the amount of foam produced when filling the spray tank |
| Pour and tap bulk density | To provide information for packaging, transport, and application |
| Pourability | To ensure that formulations have characteristics that will enable them to pour readily from containers |
| Stability at elevated temperatures | To ensure that the properties of formulations are not adversely affected by storage at high temperature |
| Storage stability at 0°C | To ensure that the properties of formulations are not adversely affected by storage during cold periods |
| Suspensibility | To ensure that a sufficient amount of active substance is homogeneously dispersed in suspension in the spray liquid |
| Volatility | To ensure that application of ultralow volume formulations does not lead to unacceptable drift due to too rapid evaporation of the sprayed droplets |
| Wettability | To ensure that certain formulations are rapidly wetted when mixed with water |
| Wet sieve test | To restrict the content of insoluble particles of sizes which could cause blockage of sprayer nozzles or filters |

Source: From Ref.^[9].

market comply with the quality specification given in the authorization, with particular emphasis on their packaging and labeling. The level of government's involvement in the quality control of the marketed pesticides varies between countries according to legislation, traditional industry-government relationships, capability, and resources.

Sampling

The objective of the sampling procedures is to provide sufficient representative material for testing the packaging, physical, and chemical properties of pesticides. As the content of each individual container should meet the quality criteria, the sample should normally be

obtained from a single packing unit. Samples should be collected safely and at an appropriate stage of production or distribution, and they should arrive intact at their destination. The FAO Guidelines for sampling procedures^[4] used for commercial or regulatory purposes are widely accepted internationally, but national standards/official procedures may be somewhat different.

Analytical Methods

To assure reliable and accurate results properly validated methods should be used only for official testing of the quality of pesticides. Considerable resources have been committed to elaborate and validate test methods by the manufacturers, national organizations, and at international level. The Collaborative International Pesticide Analytical Council Ltd. (CIPAC)^[5] and the Association of Official Analytical Chemists International (AOAC International)^[6] organize collaborative studies aiming to promote international agreement on methods for the determination of active ingredient content and for the determination of physicochemical properties of technical pesticides and formulations. The joint program of FAO and International Atomic Energy Agency^[7] provides assistance for adapting and validating suitable analytical procedures.

Analytical methods for the determination of impurities in the technical materials are usually developed by the manufacturers. As impurities may reveal information on the manufacturing process, these methods are considered confidential. They are available only for official testing purposes from the national registration authorities, and for the relevant impurities included in the FAO/WHO specifications from FAO or WHO.

Physical test methods have been validated by CIPAC, the American Society for Testing and Materials (ASTM),^[8] or, in certain cases, by the Organization for Economic Cooperation and Development (OECD), and the European Community. These methods may be regarded as definitive as, in many cases, the physical property is defined by the method of measurement.

The most comprehensive collection of validated methods is published by CIPAC.^[9] The AOAC International, the U.S. EPA, and several national agencies also publish methods for the determination of the active ingredients in pesticide formulations.

Control of Labeling

A good label should deliver all information and instructions that the users have to observe in order to

apply the pesticides safely and efficiently. This includes, among others, the statement of nominal concentration of active ingredient(s) preferably expressed in g/kg, although some national registrations allow the use of g/L or percent by weight as well.

Therefore the approval of the content of the label is normally part of the registration procedure and regulated at the national level. The general requirements are specified in the FAO International Code of Conduct on the Distribution and Use of Pesticides.^[10]

CONCLUSION

The pesticide formulation enables the delivery of the active ingredient(s) to the target object. The active ingredient content and the physicochemical properties of the formulation may significantly influence the biological activity and efficiency of the product, and the properties of formulations play an important role in the reduction of the exposure of non-target objects to pesticides and in the protection of the environment.

The quality control of pesticide formulations has its important role in ensuring that each pesticide product is adequate for the intended purpose without adverse effects on humans, animals, and the environment.

REFERENCES

1. *Manual on the Development and Use of FAO Specifications for Plant Protection Products*, 5th Ed.; FAO Plant Production and Protection Paper 149; Rome, 1999; 4, 129–140, 143–145.
2. Knowles, D.A., Ed.; *Chemistry and Technology of Agrochemical Formulations*; Kluwer Academic Publishers: Dordrecht, 1998.
3. *Catalogue of Pesticide Formulation Types and International Coding System. Technical Monograph No.2*; GIFAP: Brussels, 1989.
4. <http://www.fao.org/ag/agp/agpp/pesticid> (accessed June 2002).
5. <http://www.cipac.org> (accessed June 2002).
6. <http://www.aoac.org> (accessed June 2002).
7. <http://www.iaea.org/trc> (accessed June 2002).
8. <http://www.atmi.org> (accessed March 2002).
9. Dobrat, W.; Martijn, A.E. *CIPAC Handbook*; Collaborative International Pesticides Analytical Council Ltd.: Cambridge, 1995; Vol. F.
10. *International Code of Conduct on the Distribution and Use of Pesticides*; FAO: Rome, 1990.

Reducing Pesticide Use: Successes

David Pimentel
Maria V. Cilveti

*Department of Entomology, College of Agriculture and Life Sciences Cornell University,
Ithaca, New York, U.S.A.*

INTRODUCTION

Many countries have stated that they are implementing integrated pest management (IPM) programs. In most cases in these countries, such as the United States, there has been a slight reduction in pesticide use in a few crops, but some of this reduction has resulted from using more toxic pesticides that require grams applied per hectare instead of kilograms per hectare. For example, DDT and related chlorinated insecticides were applied at 1–2 kg/ha whereas temick and related compounds are applied at only 10 g/ha. Pesticide use in the United States, despite a few reductions in a few crops, has slightly increased over the past decade.

The prime advantage of the new, highly toxic insecticides and other related pesticides, e.g., SEVIN, is that they do not persist in the environment. Reducing the persistence of pesticides in the environment offers many advantages in terms of public health and the environment; however, the public health and environmental threat from the new highly toxic pesticides remain a major problem. For example, the World Health Organization (WHO)^[1] reports that about 26 million people are poisoned each year, with about 2,220,000 deaths. In addition to humans, other non-target species continue to suffer from the use of pesticides. For example, there are an estimated 72 million birds killed in the United States each year. Plus there are numerous other non-target species affected by pesticides.^[2,3]

OVERVIEW

Relatively few nations have made a major effort to reduce pesticide use in agriculture. The best example, and the most successful nation in reducing pesticide use, is Sweden. In 1986, the government of Sweden implemented a policy to reduce pesticide use by 50% over a 5-year period.^[4] Sweden was successful in its first effort to reduce pesticide use by 50%; then, in 1992, Sweden passed legislation to reduce pesticide use another 50%. Although Sweden has not reduced pesticide yet by 75%, they have reduced pesticide use by 68%.^[5] Associated with this 68% pesticide reduction

has been a 77% reduction in human pesticide-related health problems.

Sweden accomplished its goal of reducing pesticide use by 68% by implementing several techniques.^[4] First, the government invested in increased numbers of extension advisors and scientific investigators. The reduction in pesticide use was accomplished in part by switching from pesticides that were applied at kilogram dosages per hectare to the new highly toxic pesticides that are applied at gram dosages per hectare.

At the same time, in Sweden, changes were made in the application of pesticides in agriculture. First, pesticides were no longer applied on a routine program, whether pests were a serious problem or not. The farmers and extension workers carefully monitored the pest and beneficial organism populations to determine if there was a pest problem that warranted treatment in terms of economics and the environment.

In addition to monitoring pest and natural-enemy populations, various environmentally sound programs were implemented. These included adding crop rotations and planting crops that were relatively resistant to insect and plant-pathogen pests.

Another policy that was implemented was adjusting pesticide-use dosages that tended to kill the pests while leaving the natural-enemy population relatively unharmed. For example, in Norway, Edland^[6] found that if a lower dosage is used for certain insecticides, i.e., one-fifth the dosage recommended by the manufacturer, orchard pest control was significantly better than the manufacturer's recommended higher dosage.

When the manufacturer was asked to lower their recommended dosage, they refused at first. Finally, after numerous requests and refusals, the Norwegian government reported to the manufacturer that if they desire to continue selling pesticides in Norway, they would have to change their recommendations. Based on this warning, the manufacturer changed their recommendations to that suggested by Edland.

Another highly successful program to reduce pesticide use occurred in Indonesia. The Indonesian Government appointed a new Minister of Agriculture in 1980 and he favored the heavy use of pesticides in rice production. Under his policy, pesticide use on rice

dramatically increased. In about 4 years time, the rice farmers found that they were having trouble controlling the brown-plant-hopper pest (BPH) and rice yields were declining in many parts of Indonesia. In fact, by 1985, many thousands of hectares of rice had to be abandoned because of severe outbreaks of the BPH.

Thus instead of Indonesia being a net exporter of rice, the nation had to import rice. With rice yields declining, the President of Indonesia consulted Dr. I.N. Oka for a solution to the serious problem. Dr. Oka advised the President that the government should ban 67 of 74 pesticides in current use and implement a new pest management program. The President of Indonesia went on TV and banned 67 of 74 pesticides and announced that new policies in rice pest control would be implemented. The Minister of Agriculture was fired and Dr. Oka was placed in charge of all pest control in Indonesia.

Because of the dramatic changes in pesticide use, threats were made to Dr. Oka's life. The President of Indonesia provided bodyguards for Dr. Oka and his family. In addition, Indonesia obtained loans and grants from the World Bank and the Food and Agricultural Organization (FAO) of the United Nations to hire about 2000 new extension workers to implement Dr. Oka's policies.

Dr. Oka was an expert scientist on rice pests and their natural enemies. Thus farmers were instructed on how to identify the pest insects and beneficial insects and other arthropods. They were also instructed when pest insect populations would be a threat to rice yields and when to treat. The farmers were also instructed how to treat with pesticides to leave as many natural enemies surviving as possible.

Another important policy that Dr. Oka implemented was leaving all the rice fields in Indonesia fallow without rice for about 3 months during the year. Without rice, several of the major insect pests, especially the BPH, significantly declined, so that when rice was planted the next season, there were very few insect pests present to attack the newly planted rice.

Under Dr. Oka's policies, pesticide use was reduced by more than 65% and rice yields increased 12%—a phenomenal accomplishment! Thus the farmers benefited in terms of economics and the public benefited in terms of health and the environment.

The province of Ontario, Canada, also decided to implement a program to reduce pesticide use by 50% over a 15-year period^[7] starting in 1987. Ontario's program was similar to that in Sweden in that they replaced some of the heavy-use, high-dosage pesticides with low-dosage, highly toxic pesticides. In addition, they added extension workers and increased their investment in research on non-chemical controls.

This major effort paid off, and Ontario was able to reduce pesticide use by 50%. A survey of the farmers in Ontario found that they were highly supportive of the

goal to reduce pesticide use by 50%. The reasons that the farmers supported the program were: 1) The farmers were applying the toxic pesticides and thus were exposing themselves and their families to toxic pesticides; 2) reducing pesticide use in crop production improved farmer profits; and 3) the farmers were in favor of protecting the environment.

CONCLUSION

A detailed assessment of the potential to reduce pesticide use in the United States was conducted in the early 1900s.^[8] The investigation documented that U.S. pesticide use could be reduced by 50% if the U.S. government implemented a program similar to that of Sweden, Indonesia, and the province of Ontario. Such a program would save farmers' money, protect public health, and protect the environment.^[8]

REFERENCES

1. WHO. *Our Planet, Our Health: Report of the WHO Commission on Health and Environment*; World Health Organization: Geneva, 1992.
2. Pimentel, D.; Acquay, H.; Biltonen, M.; Rice, P.; Silva, M.; Nelson, J.; Lipner, V.; Giordano, S.; Horowitz, A.; D'Amore, M. Assessment of environmental and economic costs of pesticide use. In *The Pesticide Question: Environment, Economics and Ethics*; Pimentel, D., Lehman, H., Eds.; Chapman and Hall: New York, 1993; 47–84.
3. Pimentel, D. Ecological effects of pesticides on public health, birds and other organisms. *Reflections* **2002**, 27–28.
4. Pettersson, O. Pesticide use in Swedish agriculture: the case of a 75% reduction. In *Techniques for Reducing Pesticide Use: Economic and Environmental Benefits*; Pimentel, D., Ed.; John Wiley and Sons Ltd.: Chichester, U.K., 1997; 79–102.
5. Pesticide action network (PAN). *Pestic. Inf. Update* **2001**, 44.
6. Edland, T. Benefits of minimum pesticide use in insect and mite control in orchards. In *Techniques for Reducing Pesticide Use: Economic and Environmental Benefits*; Pimentel, D., Ed.; John Wiley and Sons Ltd.: Chichester, U.K., 1997; 197–220.
7. Surgeoner, G.A.; Roberts, W. Reducing pesticide use by 50% in the province of Ontario: challenges and progress. In *The Pesticide Question: Environment, Economics and Ethics*; Pimentel, D., Lehman, H., Eds.; Chapman and Hall: New York, 1993; 206–222.
8. Pimentel, D.; McLaughlin, L.; Zepp, A.; Lakitan, B.; Kraus, T.; Kleinman, P.; Vancini, F.; Roach, W.J.; Graap, E.; Keeton, W.S.; Selig, G. Environmental and economic impacts of reducing U.S. agricultural pesticide use. *BioScience* **1991**, 41, 402–409.

Resistance to *Bt* Transgenic Plants

Hugo Cerda-Perez

Simon Rodriguez University, Caracas, Venezuela

Denis J. Wright

Department of Biological Sciences, Imperial College London, Ascot, U.K.

INTRODUCTION

Microbial insecticides based on *Bacillus thuringiensis* (*Bt*) can be effective control agents because of their activity and high degree of specificity. Recent developments in biotechnology have resulted in transgenic crops expressing *Bt* crystal (Cry) toxins^[1] with, in some cases, a reduction in the use of conventional insecticides.^[2] With such success comes the danger that prolonged and uniform exposure to *Bt* crops will intensively select for adaptation to Cry toxins in pest populations.^[3,4] In such circumstances, microbial *Bt* products that have been utilized successfully in integrated pest management programs and in organic farming could also become ineffective.^[5,6]

TRANSGENIC INSECTICIDAL CULTIVARS

Bt produces the following various proteins with insecticidal activity: 1) α -exotoxin (heat-labile); 2) β -exotoxin (heat-stable); 3) δ -endotoxin (Cry); and 4) louse-factor, the Cry toxins being the most important. Different *Bt* isolates contain different combinations of Cry toxins. For example, *Bt* subsp. *kurstaki* HD-1 contains genes that code for at least five Cry toxins: Cry1Aa, Cry1Ab, Cry1Ac, Cry2Aa, and Cry2B.^[3]

The mode of action of *Bt* is on the insect midgut cell membrane. In the bacterium, Cry toxins are produced as parasporal crystalline inclusions. In susceptible insects, these inclusions are dissolved in the midgut, releasing protoxins (which range in size from 27 to 140 kDa) that are proteolytically converted into smaller, toxic polypeptides. There is extensive variation in the size and structure of the inclusion proteins, the protoxins, and the active Cry toxins, which is presumed to relate to their specificity.^[7,8]

Following activation, Cry toxins bind to specific receptors on the midgut epithelium; these receptors are thought to be aminopeptidases.^[9] Different domains of the toxin molecule are responsible for receptor recognition and pore formation. In Cry1 toxins, the C-terminal domain is involved in specificity (binding) and the N-terminal domain in toxicity (pore formation).^[10]

After binding to the midgut epithelia cells, the Cry toxins generate pores in the cell membrane, disturbing cellular osmotic balance and causing the cell to swell and lyse (termed “colloid-osmotic lysis”).

Since 1996, millions of acres have been planted with *Bt* transgenic cotton, maize, and potato that express a Cry toxin. The introduction of other crops expressing Cry toxins is imminent and the overall acreage of *Bt* crops is expected to continue to increase, particularly in developing countries. The global market value for transgenic crops is expected to increase from <\$500 million in 1996 to \$20 billion by 2010.^[4]

For many years, the likelihood of resistance to Cry toxins was considered remote because of the very complex mode of action involving multiple toxins and multiple target sites.^[3] However, where *Bt* sprays have been intense, in high-value crucifer production, resistance to *Bt* has been shown to develop on a wide scale in the diamondback moth, *Plutella xylostella*.^[11] With the advent of *Bt* transgenic crops, the selection pressure for resistance will be greatly increased for a number of crop pests, particularly in cotton, maize, potato, and vegetables.

THE GENETICS AND MECHANISMS OF RESISTANCE

A number of studies with field-derived populations of *P. xylostella* resistant to *Bt* have shown that resistance is inherited as a recessive trait.^[11] Resistance in some other populations of *P. xylostella* has been found to be incompletely recessive or incompletely dominant.^[12] Huang et al.^[13] also showed that resistance in a laboratory-selected population of the European corn borer, *Ostrinia nubilalis*, was incompletely dominant.

The majority of the studies of the mechanism of *Bt* resistance appear to be related to loss of membrane binding by Cry toxins. However, the various steps involved in the mechanism of *Bt* toxicity provide other opportunities for resistance mechanisms. Evidence for a second mechanism for resistance to Cry toxins has been reported in laboratory-selected populations of the Indianmeal moth, *Plodia interpunctella*, which

is associated with the loss of a major midgut protease that activates Cry protoxins.^[14]

RESISTANCE MANAGEMENT STRATEGIES

Various strategies to manage resistance to *Bt* transgenic crops toxins have been proposed,^[3,4,15] all of which assume that the frequency of resistance alleles will decrease when the selection pressure is reduced or discontinued: 1) mixtures, mosaics or rotations of transgenic plants; 2) time- or tissue-specific expression of toxin; 3) low doses of toxin in combination with natural enemies; 4) coexpression of different *cry* genes; and 5) high expression (dose) with refugia, which is the strategy being recommended currently.^[15]

The high-dose refugia strategy is based on three assumptions:

1. That resistance will be inherited as a recessive trait. That is, the majority of the heterozygous progeny will be disabled or killed at the same dose as for homozygous susceptible larvae, thereby limiting the spread of the resistance alleles in the population.
2. That insects with resistance to *Bt* will be initially rare, and will almost always mate (at random) with susceptible wild-type insects, giving rise to heterozygous progeny.
3. That resistant individuals will be at a competitive disadvantage because of a fitness cost incurred by carrying the resistance allele.

If recessive inheritance, randomized mating, and tightly linked population dynamics of insects that develop on *Bt* and non-*Bt* plants are assumed, the effective size of refugia can be estimated as follows:

$$\text{Refuge \%} = \frac{A}{(A + B/q^2)} \times 100\%$$

where *A* is the number of susceptible adult insects produced in the refuge in a specified generation; *B* is the number of resistant homozygotes; and *q* is the frequency of resistance alleles.^[15]

Recent studies have provided evidence to question, to some degree, these assumptions. As mentioned above, some studies have shown resistance to *Bt* to be incompletely recessive or incompletely dominant. Meanwhile, Darby^[16] and Liu et al.^[17] found evidence of developmental asynchrony in *Bt*-resistant populations of *P. xylostella* and the pink bollworm, *Pectinophora gossypiella*, respectively, on crops with Cry1Ac compared with non-*Bt* crops—a phenomena that could lead

to non-random mating. Tabashnik et al.^[18] showed that up to 94% of *P. gossypiella* males dispersed 400 m or less from the release sites in transgenic cotton crops. They concluded that this movement was not sufficient to distribute wild males randomly between *Bt* and non-*Bt* cotton. The assumption on fitness costs has also been questioned in one study, where the larvae of *P. xylostella* that had evolved resistance to foliar spray of *Bt* subsp. *kurstaki* in the field showed no apparent fitness costs.^[19]

However, a recent field study has shown that the frequency of *Bt* resistance in *P. gossypiella* collected from cotton fields in Arizona actually declined from 1997 to 1999, despite the introduction of *Bt* cotton.^[20] Shelton et al.^[21] have reported the first detailed field experiments to assess the effectiveness of different refugia strategies with a *Bt* crop.

CONCLUSION

Recent studies have shown that some populations of insects may not show the genetic and biological characteristics assumed to be a prerequisite for the high-dose refugia strategy to succeed. In this context, it is the way in which refugia are used that would appear to be central to resistance management. The presence of resistant heterozygotes in some insect populations possessing some degree of tolerance to *Bt* toxins also suggests that high levels of expression of toxins need to be maintained during the crop cycle. In the future, given the greater number of insect pests that will be exposed to Cry toxins in transgenic crops and the different ecological conditions that will exist, many scenarios for the evolution of resistance may occur and a pest-crop by pest-crop approach should be taken to optimize the resistance management strategy.

ACKNOWLEDGMENTS

Hugo Cerda was supported by a Ph.D. fellowship from CONICT Venezuela.

REFERENCES

1. Mellon, M., Rissler, J., Eds.; *Now or Never: Serious Plants to Save a Natural Pest Control*; Union of Concerned Scientist Publications: Cambridge, MA, 1998; 149.
2. Fitt, G.P. An Australian approach to IPM in cotton: Integrating new technologies to minimise insecticide dependence. *Crop Prot.* **2000**, *19*, 793–800.

3. Whalon, M.E.; McGaughey, W.H. *Bacillus thuringiensis*: use and resistance management. In *Insecticidal with Novel Modes of Action: Mechanism and Application*; Ishaaya, I., Degheele, D., Eds.; Springer Verlag: Berlin, 1998; 106–137.
4. Frutos, R.; Rang, C.; Royer, M. Managing insect resistance to plants producing *Bacillus thuringiensis* toxins. *Crit. Rev. Biotechnol.* **1999**, *19*, 227–276.
5. Paoletti, M.G.; Pimentel, D. Genetic engineering in agriculture and the environment. *Bioscience* **1996**, *46*, 665–673.
6. Paoletti, M.G.; Pimentel, D. Environmental risk of pesticides versus genetic engineering for agricultural pest control. *J. Agric. Environ. Ethics* **2000**, *12* (3), 279–303.
7. Estada, U.; Ferré, J. Binding of insecticidal crystal proteins of *Bacillus thuringiensis* to the midgut brush border of the cabbage looper *Trichoplusia ni* (Lepi: Noctuidae) and selected for resistance to one of the crystal protein. *Appl. Environ. Microbiol.* **1994**, *60*, 3840–3846.
8. Feldmann, F.; Dullemans, A.; Waalwijk, C. Binding of the CryIV D toxins of *Bacillus thuringiensis* subsp. *israelensis* to larval dipteran midgut proteins. *Appl. Environ. Microbiol.* **1995**, *61*, 2601–2605.
9. Burton, S.L.; Ellar, D.J.; Li, J.; Derbyshire, D.J. *N*-Acetylgalactosamine on the putative insect receptor aminopeptidase N is recognised by a site on the domain III lecitin-like fold of a *Bacillus thuringiensis* insecticidal toxin. *J. Mol. Biol.* **1999**, *287*, 1011–1022.
10. Frankenhuyzen, K.V. The challenge of *Bacillus thuringiensis*. In *Bacillus Thuringiensis: An Environmental Biopesticide: Theory and Practice*; Entwistle, P.F., Cory, J.S., Bailey, M.J., Higgs, S., Eds.; John Wiley and Sons: Chichester, 1993; 1–36.
11. Tabashnik, B.E.; Liu, Y.B.; Malvar, T.; Heckel, D.G.; Masson, L.; Ballester, V.; Granero, F.; Mensua, J.L.; Ferré, J. Global variation in the genetic and biochemical basis of diamondback moth resistance to *Bacillus thuringiensis*. *Proc. Natl. Acad. Sci. U. S. A.* **1997**, *94*, 12,780–12,785.
12. Sayyed, A.H.; Haward, R.; Herrero, S.; Ferré, J.; Wright, D.J. Genetic and biochemical approach for characterization of resistance to *Bacillus thuringiensis* toxin Cry1Ac in a field population of the diamondback moth, *Plutella xylostella*. *Appl. Environ. Microbiol.* **2000**, *66*, 1509–1516.
13. Huang, F.; Buschman, L.L.; Higgins, R.A.; McGaughey, W.H. Inheritance of resistance to *Bacillus thuringiensis* toxin (Dipel ES) in the European corn borer. *Science* **1999**, *284*, 965–967.
14. Oppert, B.; Kramer, K.J.; Beeman, R.W.; Johnson, D.; McGaughey, W.H. Proteinase-mediated insect resistance to *Bacillus thuringiensis* toxins. *J. Biol. Chem.* **1997**, *272*, 23,473–23,476.
15. Gould, F. Sustainability of transgenic insecticidal cultivars: integrating pest genetic and ecology. *Annu. Rev. Entomol.* **1998**, *43*, 701–726.
16. Darby, A.C. Can developmental time differences in transgenic crops and their refugia result in assortative mating between resistant insects?. In *M.Sc. Thesis*, Imperials College of Science, Technology and Medicine: Silwood Park, Ascot, 1998; 85 pp.
17. Liu, Y.; Tabashnik, B.E.; Dennehy, T.J.; Patin, A.L.; Bartlett, A.C. Development time and resistance to Bt crops. *Nature* **1999**, *400*, 519 pp.
18. Tabashnik, B.E.; Patin, A.L.; Dennehy, T.J.; Liu, Y.B.; Miller, E.; Staten, R.T. Dispersal of pink bollworm (Lepidoptera: Gelechiidae) males in transgenic cotton that produces a *Bacillus thuringiensis* toxin. *J. Econ. Entomol.* **1999**, *92*, 772–778.
19. Tang, J.D.; Collins, H.L.; Roush, R.T.; Metz, T.D.; Earle, E.D.; Shelton, A.M. Survival, weight gain, and oviposition of resistant and susceptible *Plutella xylostella* (Lepidoptera: Plutellidae) on broccoli expressing Cry1Ac toxin of *Bacillus thuringiensis*. *J. Econ. Entomol.* **1999**, *92*, 47–55.
20. Tabashnik, B.E.; Patin, A.L.; Dennehy, T.L.; Liu, Y.-B.; Carrière, Y.; Sims, M.A.; Antilla, L. Frequency of resistance to *Bacillus thuringiensis* in field populations of pink bollworm. *Proc. Natl. Acad. Sci. U. S. A.* **2000**, *97*, 12,980–12,984.
21. Shelton, A.M.; Tang, J.D.; Roush, R.T.; Metz, T.D.; Earle, E.D. Field test on managing resistance to *Bt*-engineered plants. *Nat. Biotechnol.* **2000**, *18*, 339–342.

Rice Diseases: Ecology and Control

T. S. Thind

Vineet K. Sharma

Department of Plant Pathology, Punjab Agricultural University,
Ludhiana, Punjab, India

INTRODUCTION

Rice (*Oryza sativa* L.) is grown under varying climatic conditions in the tropical and subtropical regions of the world. Diseases are serious constraints affecting rice yield and quality. Several major diseases, caused by different pathogens, occur on this crop, resulting in significant damage to the grain and straw yield. Diseases are estimated to cause annual yield and quality losses of 8%–10% in rice, which may go up to 50% in severe cases.^[1]

Rice diseases are affected by several environmental factors, cultural practices, and the varieties cultivated in a particular region. The adoption of short-statured, high-yielding varieties (semi-dwarfs) that have high nitrogen fertilizer requirements has also contributed to increased losses from diseases. Rice diseases can be grouped into different categories based on the plant parts infected (Table 1). Although some diseases including blast, brown spot, bacterial blight, and sheath blight can be placed in two or more categories, these categories, in general, facilitate an understanding of the damage caused by these diseases and aid in identifying diseases. The adoption of various control methods involving resistant varieties, cultural practices, and chemicals can help in managing rice diseases.

EFFECT OF DISEASES ON RICE

In recent times, the impact of diseases on rice production has increased significantly. Important fungal diseases in rice include blast, brown spot, sheath blight, false smut, sheath rot, stem rot, bakanae, and kernel smut. Rice blast damages plants and causes yield reduction in a number of ways. Lesions on leaf blades reduce the effective leaf area for photosynthesis and infection of the culms at nodes causes greater damage than leaf infections. Neck infection results in the formation of half-filled and totally chaffed panicles. Losses in severely affected fields may exceed 50%. Rice plants at the tillering stage may be killed when leaves are severely infected. Infection of the upper most culm node (neck) usually causes high yield losses, as it results in unfilled grains. Brown spot disease may result in poor germination of infected seeds and lead to up to

50% of seedling mortality. It becomes important on foliage during the maximum tillering to grain formation stages. Sheath blight is an increasing concern, especially in intensified rice production involving high-yielding short or semi-dwarf varieties. Yield losses due to this disease may reach 25% if the infection spreads up onto the flag leaves. This disease starts during the maximum tillering stage of the crop and increases as the plant grows older.

False smut has become an important disease in high-yielding, nitrogen-responsive cultivars. The disease affects the early flowering stage of the rice crop, destroying the ovary. The second stage of infection occurs when the spikelet reaches maturity. Losses in the crop yield are due to the conversion of kernels into ball-like structures (pseudosclerotia) and increased sterility of the adjacent kernels. Bakanae disease or foot-rot is widely distributed and infection usually occurs during the seedling and tillering stages, with crop losses reaching 20%.

Sheath rot appears late during growing season and impacts crops from heading to maturity. It usually attacks the uppermost leaf sheath and causes the panicles to rot. Stem rot affects the rice crop during the early heading and grain filling stages. The leaf sheaths decay, causing decreased grain filling. Kernel smut, although less important economically, may damage the quality of cooked rice.

Bacterial blight and bacterial leaf streak are two important bacterial rice diseases. Yield losses due to bacterial blight correspond to the plant growth stages at which the rice plants are infected and losses are higher if the disease occurs earlier in the season. Damage is due to the partial or total blighting of leaves or the complete wilting of the affected tillers. Bacterial blight is reported to reduce annual rice production in Asia up to 60%. Bacterial leaf streak is a major rice disease in several Asian countries and is not known to occur in temperate countries, including Japan. It is usually observed during the tillering stage and losses ranging from 5 to 30% have been reported. Bacterial grain and seedling rot, caused by *B. glumae*, is rapidly becoming a major rice disease in the United States, Japan, and Korea. This disease is especially severe when night temperatures are higher than normal.^[2] In Korea, the bacterium has also been found to cause

Table 1 Major rice diseases, their symptoms, survival of pathogens and potential for damaging rice

| Disease (Pathogen) | Symptoms | Survival of pathogen | Damage potential |
|--|---|---|------------------|
| A. Diseases affecting the leaf blade | | | |
| i. Leaf blast (<i>Pyricularia grisea</i>) | Leaf spots spindle shaped with brown or reddish-brown margins, ashy centres and pointed ends | Mycelium and conidia in seed, diseased straw, collateral hosts | High |
| ii. Brown spot (<i>Bipolaris oryzae</i>) | Small round, dark brown spots with a dull yellow margin enlarging to oval spots with a gray centre and a dark brown margin | Mycelium in seed, infected plant debris, stubble, weed hosts | Low to moderate |
| iii. Sheath blight (<i>Rhizoctonia solani</i>) | Greenish gray irregular lesions on leaf sheath with dark line on margins | Sclerotia and/or mycelium in diseased plant debris and seed, several weeds as collateral hosts | High |
| iv. Leaf smut (<i>Entyloma oryzae</i>) | Short, linear to elliptical black spots on both sides of leaves | Teliospores in the leaf trash in the field | Low |
| v. Bacterial leaf blight (BLB) (<i>Xanthomonas oryzae</i> pv. <i>oryzae</i>) | Lesions begin as water-soaked stripes near the leaf tip or margin and enlarge rapidly forming yellow to straw coloured areas with wavy margins | Infected rice straw, stubbles, volunteer plants, weed hosts, rhizosphere of succeeding rice crop and seed | Moderate to high |
| vi. Kresek phase of BLB (<i>X. oryzae</i> pv. <i>oryzae</i>) | Lesions may cover the entire blade, turn white to yellow within a week after transplanting | Same as given in A (v) | High |
| vii. Bacterial leaf streak (<i>X. oryzae</i> pv. <i>oryzicola</i>) | Lesions begin as fine, water-soaked to translucent long streaks in the interveinal areas which later turn yellow or orange brown | Infected seed | Low |
| viii. Tungro (Rice tungro bacilliform virus and rice tungro spherical virus) | Leaf discoloration begins from leaf tip and extends down to the blade or the lower leaf portion. Infected leaves may also show mottled or striped appearance. On older leaves, small, rusty, necrotic spots may also develop later with discolored area | Wild rice, ratoons, weeds, stubbles, nursery bed seedlings | Low to moderate |
| B. Diseases affecting leaf sheath and stem | | | |
| i. Blast (<i>P. grisea</i>) | Internodal infection of the culm occurs in a banded pattern. Grayish black or dark necrotic lesions appear on culm. Nodal infection causes the culm to break at the infected node. The grains are mostly half filled or unfilled | Same as given in A (i) | High |
| ii. Sheath blight (<i>R. solani</i>) | Sheath with large greenish gray or dark edged whitish lesions which are oval, oblong, cobra patches, irregular and necrotic. Small, white or brown, globular sclerotial bodies are loosely attached to the surface | Same as given in A (iii) | High |

(Continued)

Table 1 Major rice diseases, their symptoms, survival of pathogens and potential for damaging rice (*Continued*)

| Disease (Pathogen) | Symptoms | Survival of pathogen | Damage potential |
|--|--|---|------------------|
| iii. Stem rot (<i>Sclerotium oryzae</i>) | Small, black linear lesions appear on the outer sheath near water level which later turn to dark brown discoloration. Leaves of affected sheaths die. Infected stem rots and breaks over. Black blotches are noted on culms under rotted sheaths. Numerous small, black sclerotia appear within sheaths and stems | Sclerotia in soil, old rice straw, stubbles | Low to moderate |
| iv. Sheath rot (<i>Sarocladium oryzae</i>) | Brown to reddish brown areas with irregular outlines, entire leaf sheath converted from brown to straw colour. Panicles may be twisted, covered with white powdery mass, and have florets with brown discoloration. Infected panicles remain sterile, shriveled, or with partially filled grains | Seed-borne, infected plant debris, wild rice, grasses | Low to moderate |
| v. Bakanae or foot rot (<i>Fusarium moniliforme</i>) | Leaves and leaf sheaths dry up, lower nodes are discolored and sometimes with adventitious roots, pink bloom on sheath above water level and infected plants are generally taller than normal | Seed and soil-borne | Low to moderate |
| vi. Tungro (Rice tungro bacilliform virus and rice tungro spherical virus) | Entire plant with typical orange yellow color with chlorosis, dwarfed, non-tillering and ultimately collapsing | Same as given in A (viii) | Low to moderate |
| C. Diseases affecting panicles, florets, and grains | | | |
| i. Rotten neck and panicle blast (<i>P. grisea</i>) | Infection of panicle base causes neck rot. Lesions on the neck are grayish brown, cause the girdling of the neck and the panicles fall over. Lesions on the branches of the panicles and on the spikelet pedicels are brown to dark brown, later turning gray. Portion of branch above these lesions turns white and grains stop filling. Grains partially chaffy, brittle or unfilled | Same as given in A (i) | High |
| ii. Brown spot (<i>Bipolaris oryzae</i>) | Dark brown or black, oval or oblong spots on grain hulls often large enough to cover entire grain. Affected grains may be poorly filled and chalky | Same as given in A (ii) | Low to moderate |
| iii. False smut (<i>Ustilagoidea virens</i>) | Individual rice grain completely replaced by large spherical yellowish or orange body which changes to powdery mass later | Sclerotia in plant debris in soil | Low to moderate |

| | | | |
|---|---|---|------------------|
| iv. Kernel smut (<i>Neovossia horrida</i>) | Grain poorly filled and covered with and/or partially filled with a black, powdery mass that rubs off quickly | Teliospores fallen on the soil or present on the seed | Moderate |
| v. Sheath blight (<i>R. solani</i>) | Panicle emerging from boot, fails to branch out, turns paper white with some brown discoloration, remains upright and produces poorly filled or empty grains | Same as given in A (iii) | High |
| vi. Sheath rot (<i>Sarocladium oryzae</i>) | Severe infection causes entire or parts of young panicles to remain within the sheath. Unemerged panicles rot and florets turn red-brown to dark brown. An abundant whitish powdery growth appears inside affected sheaths and young panicles. Most of the grains are discolored, sterile, shriveled, partially or unfilled | Same as given in B (iv) | Low to moderate |
| vii. Bacterial grain rot (<i>Burkholderia glumae</i>) | The color of spikelets is initially changed to grayish-brown and then straw color and panicles remain erect and get blighted. The bacterium causes grain rotting when temperature and moisture are high | Infected seeds | Low to moderate |
| D. Diseases affecting the entire plant | | | |
| i. Bacterial leaf blight (<i>X. oryzae</i> pv. <i>oryzae</i>) | Infected leaves turn yellow, dry rapidly and wither. Blighting extends to the leaf sheath and culms, kills the tiller or whole culm. In kresak phase entire plant wilts completely. Bacterial ooze appears at the cut ends of the plant | Same as given in A (v) | Moderate to high |
| ii. Bakanae or foot rot (<i>Fusarium moniliforme</i>) | Plants pale to yellowish green and thin, abnormally elongated or rotting in patches in the field, roots also rot | Same as given in B (v) | Low to moderate |
| iii. Tungro (Rice tungro bacilliform virus and rice tungro spherical virus) | Brownish yellow in coloration and severe to mild stunting, reduced tillering, leaves tested with iodine show black or deep brown colour | Same as given in A (viii) | Low to moderate |
| iv. Bacterial seedling rot (<i>Burkholderia glumae</i>) | There is seedling rot in rice nursery boxes | Same as given in C (vii) | Low to moderate |

wilt in other crops, such as tomato, eggplant, and pepper.

Among viral rice diseases, tungro is the most devastating throughout the world. It affects all growth stages of rice, but the early growth (vegetative) stage is more prone to attack, causing yield losses as high as 100% in severe cases.

DISEASE DEVELOPMENT

Rice crop, in general, needs a hot and humid climate and is best suited to regions that have high humidity, high rainfall, and temperatures ranging from 21 to 37°C. Such growing conditions also favor the development of major diseases.

To understand the development of rice diseases, knowledge about their primary source of inoculum and pathogen carryover and dissemination is essential. Several pathogens survive in the seed, while others survive in the infected crop debris/residues left after harvest or on the collateral weed hosts (Table 1).

Diseases like blast, brown spot, bacterial blight, false smut, kernel smut, bacterial grain and seedling rot, and sheath blight are favored by a combination of high atmospheric humidity, cloudy and rainy days, longer dew duration, and moderate to high temperatures. While low to moderate temperature (15°C–25°C) favors the development of blast and brown spot, diseases such as bacterial blight, bacterial leaf streak, kernel smut, sheath blight, and foot rot develop and spread more rapidly at relatively higher temperatures ranging from 25 to 35°C.^[3]

The application of heavy doses of nitrogen makes the rice crop more prone to attack by several diseases except for a few, including brown spot, which are more serious in nitrogen-deficient, light-textured soils. The spread of tungro, a viral disease, occurs through its leaf hopper vector *Nephotettix virescens*.^[4] Stem borer infestation results in increasing incidences of stem rot and sheath rot. Late maturing varieties are more susceptible to false smut, while early varieties of rice suffer more from kernel smut. Likewise, the young crop stage (less than three weeks old) is more susceptible to bacterial blight, leading to the development of the kresak phase. The incidences of blast and kernel smut are increased if the crop is grown in light-textured soils. Shaded conditions aggravate diseases like bacterial blight and bacterial leaf streak.

DISEASE CONTROL OPTIONS

Improved production practices suited to a particular region and good growing conditions increase plant vigor and are likely to reduce the chances of plants

succumbing to most rice pathogens. The integration of various methods, such as host resistance, cultural practices, and need-based chemical use, provides better disease management. Only certified seeds of recommended varieties in a particular region should be used since seed serves as a source of primary inoculum for several rice pathogens.

Cultural Practices

High doses of nitrogenous fertilizers should be avoided, as they make plants more succulent—facilitating the attack of several fungal and bacterial pathogens. Nitrogen should be applied in small increments based on actual crop requirements. Close planting should be avoided as it encourages the spread of diseases like bacterial leaf blight and sheath blight. The removal and destruction of straw and stubble from the infected rice crop after harvest reduces the inoculum levels of pathogens like *R. solani* (sheath blight), *F. moniliforme* (bakanae), *S. oryzae* (stem rot), *S. oryzae* (sheath rot), *E. oryzae* (leaf smut), and *X. oryzae* pv. *oryzae* (bacterial blight) for the succeeding crop. A dry heat treatment of rice seed can eradicate grain rot bacterium, *B. glumae*.

Grass weeds in and around rice fields serve as collateral hosts for many rice pathogens. The removal and destruction of these weed hosts and volunteer rice seedlings help in reducing the inoculum levels of several rice pathogens, notably *R. solani*, *X. oryzae* pv. *oryzae*, *P. grisea*, *B. oryzae*, and tungro virus. Transfer of water from infected fields to adjacent rice fields should be avoided, especially in the case of bacterial leaf blight.^[5]

Resistant Varieties

The use of resistant varieties is the most effective, economical, and common management practice adopted by farmers in most rice growing countries. This is particularly true for the control of diseases like bacterial blight, tungro, blast, kernel smut, leaf smut, and false smut. When different strains of pathogens are present, notably in *X. oryzae* pv. *oryzae* and *P. grisea*, it is advisable to grow varieties possessing field resistance genes.^[3,6]

The genetic transformation of rice offers opportunities for the improvement of existing elite varieties. It allows breeders to develop new varieties through the introduction of cloned genes, which have resistance to important diseases, into commercial varieties. The transformation of elite indica rice variety IR72 with the *Xa21* gene has been reported to confer resistance to bacterial blight pathogen and this resistance was shown to be stably inherited in subsequent generations.^[7]

Chemical Control

Several chemicals are now available for the effective control of diseases where host resistance is unstable or lacking. The seed should be properly treated with suitable fungicides/antibiotics before sowing. Seed treatment with carbendazim or triazole fungicides and antibiotics like streptomycin is effective in significantly reducing initial inoculum levels of several fungal and bacterial pathogens. Foliar sprays at the tillering and booting stages with carbendazim, tricyclazole, propiconazole, copper oxychloride, mancozeb, flusilazole, tebuconazole, and the recently developed strobilurin compounds^[8] are effective in managing most commonly occurring fungal diseases. The chemical control of insect vectors (leaf hoppers) with insecticides like cypermethrin and carbofuran is important in checking further spread of the virus causing tungro.

CONCLUSION

Rice diseases are an important constraint in realizing desirable yield levels. The adoption of high-yielding nitrogen-responsive varieties has increased the incidence of diseases. Rice is cultivated mostly in subtropical to tropical regions, and these warm and humid growing conditions favor the development of many diseases. The cultivation of varieties with stable and multiple disease resistance genes and the adoption of improved cultural practices, along with need-based

chemical use, help in effectively managing rice diseases. Advanced molecular techniques can be helpful in the identification and introduction of resistance genes from a wide range of alien sources.

REFERENCES

1. Singh, C. *Modern Techniques of Raising Field Crops*; Oxford and IBH Publishing Co. Pvt. Ltd.: New Delhi, 1983.
2. Jeong, Y.; Kim, J.; Kim, S.; Kang, Y.; Nagamatsu, T.; Hwang, I. Toxoflavin produced by *Burkholderia glumae* causing rice grain rot is responsible for inducing bacterial wilt in many field crops. *Plant Dis.* **2003**, *87* (8), 890–895.
3. Ou, S.H. *Rice Diseases*, 2nd Ed.; Commonwealth Mycological Institute: UK, 1985.
4. Hibino, H. Biology and epidemiology of rice viruses. *Annu. Rev. Phytopathol.* **1996**, *34*, 249–272.
5. Padhi, B.; Gangopadhyay, S. Diseases of rice and their management. In *Diseases of Field Crops and Their Management*; Thind, T.S., Ed.; National Agricultural Technology Information Centre: Ludhiana, India, 1998; 35–52.
6. Thind, B.S. Bacterial blight of rice—an overview. *Plant Dis. Res.* **2002**, *17* (2), 227–251.
7. Tu, J.; Ona, I.; Zhang, Q.; Mew, T.W.; Khush, G.S.; Data, S.K. Transgenic rice variety 'IR72' with *Xa21* is resistant to bacterial blight. *Theor. Appl. Genet.* **1998**, *97*, 31–36.
8. Hewitt, H.G. *Fungicides in Crop Protection*; CAB International: Wallingford, U.K., 1998.

Rodent Exclusion

Krishnoji Rao Muktha Bai

Food Protectants and Infestation Control Department, Central Food Technological Research Institute, Mysore, Karnataka, India

INTRODUCTION

Rodents are important and dangerous vertebrate pests of humans, and they are ubiquitous and abundant all over the world. There are more than 4600 species of mammals in the world, of which 1800 are rodents (about 150 are of pest status), whereas >20–25 are of economic importance.^[1] Of these, the three important cosmopolitan, commensal species are: 1) the Norway rat (*Rattus norvegicus*, Berkenhout); 2) the roof rat (*Rattus rattus*, Linnaeus); and 3) the house mouse (*Mus musculus*, Linnaeus). However, some others developing into commensals are the lesser bandicoot rat (*Bandicota bengalensis*, Gray), the Polynesian rat (*Rattus exulans*), and the multimammate rat (*Mastomys* or *Praomys natalensis*).^[2] Two or more species are generally found in all countries of the world, and in many places, they are joined by other destructive native rodents such as the bandicoot rat (*Bandicota indica*, Bechstein) of India, Ceylon, and China; the spiny mouse (*Acomys coharinus*) of Africa; and the *Rattus flavipectus* of southern China and Indochina. Because of their close proximity to humans, they are able to spread many dreaded and infectious diseases such as plague, hantavirus pulmonary syndrome (HPS), and hemorrhagic fever with renal syndrome (HFRS), causing significant morbidity and mortality.^[3,4]

They also inflict substantial food losses of qualitative and quantitative nature. The average food loss solely because of rodents in developed countries is estimated to be 1–5%, whereas it is 2–30% in developing countries.^[5] The word *rodent* means “to gnaw,” and all rodents (rats, mice, and bandicoots) possess two pairs of sharp, chisel-like incisor teeth. Because the hardness of the incisor’s enamel is rated at 5.5 in Moh’s scale, they are able to gnaw most materials of lower hardness value with ease, which includes lead (1.5), aluminum (2.0), wood (1.5–3.0), hard rubber cork (2.5), paper (<1.0), and copper (2.5–3.0).^[6] To keep incisors in size and shape and to avoid overgrowth, they gnaw and damage various materials, structures, buildings, furniture, rubber, and hard plastics. Gnawing of insulation of electrical wires and cables often causes short circuits and fire outbreaks, besides hampering production and

causing economic losses worth millions of dollars. Despite efforts and substantial expenditures incurred in controlling these pests, effective management appears elusive but remains an important and challenging task ahead for technologists, scientists, and governments.

IS THERE A NEED TO ADOPT RODENT EXCLUSION?

The indifference and negligence of humans in handling food materials and refuse, the destruction of natural predators, and the provision of a conducive environment for breeding and propagation of rodents are the main factors that have intensified pest problems and have made control programs more intricate. Among several methods of control measures, the chemical method is the most widely used application in most parts of the world because of market availability, low cost, ease of application, immediate reduction of rodent population density, and quick relief.^[7] However, the development of resistance in most species to anticoagulants, bait shyness, and non-target hazards associated with the use of acute rodenticides have necessitated a review of methods in current use for their proper application and an exploration of the possibilities of non-chemical approaches to alleviate rodent problems. Because the primary objective of all control measures is to achieve long-lasting relief from pest problems—keeping in mind the safety of humans and less adverse effects on the environment—it is imperative to explore measures that are sustainable and eco-friendly. Moreover, with increasing public concern and awareness about environmental hazards, there has been a growing interest in techniques that can be used to reduce or replace the use of rodenticides. There is no “single” universal method that could be deployed against different rodent species under all types of environments because of inherent deficiency in each method (Table 1). In this context, the “rodent exclusion” approach, which aims to prevent rodents, rather than allow them to establish and incur considerable losses prior to control action, appears more appropriate and beneficial.

Table 1 Rodent control measures

| Control measures | Limitations | Solutions |
|--|--|---|
| Trapping | Trap shyness, cost, skill and time, live rats disposal | Improved traps effective against all species |
| Rodenticides | | |
| i) Acute | Prebating, bait shyness, hazards to non-target species, accidental poisoning | Improved bait formulations, bait stations, additives (which enhance safety to non target species) in baits |
| ii) Anticoagulants | Resistance, environmental hazards | Judicious and proper use, pulsed baiting, compounds with different modes of action (combat resistance) |
| Burrow fumigation | Lack of trained and experienced personnel, limited knowledge of burrow patterns (target species) | Newer or improved formulations with good penetration (to reach the target pest) properties and simpler devices |
| Repellents (chemical, ultrasound, electromagnetic devices) | Effects short-lived, tolerance, varied results, non-target hazards | Improved formulations and devices, area repellents, bioactive compounds, aversive agents |
| Predators (natural predators, pathogens) | Non-availability, varied effects, human attitude, uneconomical, non-specific, danger of food contamination | Breeding genetically aggressive predators, viral-vectored immunocontraception method |
| Sanitation and rodent proofing | High cost, maintenance, socio-economic and cultural factors | Rat-proof building, landscape designing, low-cost effective storage structures, rodent-proofing devices (ready to use), yields permanent results when combined with IPM |

MANAGEMENT OF RODENT PESTS BY EXCLUSION METHOD

Rodent exclusion involves mainly the physical method of reducing the impact of rodent population: by 1) lowering the carrying capacity of the habitat by sanitation and hygiene practices to discourage their living in and around human settlements; and 2) stopping or denying their entry into structures, buildings, homes, factories, or many other habitats by proofing.

Sanitation and Hygiene

This involves mainly good housekeeping and storage practices, wherein food and feed are stored properly in sealed, closed, or rodent-proof containers or rooms (for bulk stores), whereas residues, spillage, and wastes are collected in containers with lids until their disposal. Because water is vital to most rodents' survival, to prevent their access to water, it is essential to repair leaky taps, pipes, and drain spouts; close ditches; and remove places of water stagnation. Weeds, grass, and bushy shrubs that provide harborage need to be eliminated around buildings (at least 1 m), and regular maintenance is important. Although it is possible to reduce or keep in check pest populations to a great extent by

manipulating mostly sanitation aspects, lasting effects can be achieved when combined with rodent proofing.

Rodent Proofing

Because the objective of proofing is to prevent their access into buildings, it is essential to have sound knowledge of the target pests' physical abilities (Table 2). This aids in designing economical but successful rodent-proof measures needed for a particular situation. For example, although their usual entry points into buildings are through holes larger than 0.5 cm (mice) and 1.5 cm (rats), doors, windows, ventilators, air bricks, foundations, water and electrical pipes or conduits, drains, trees, cracks, and crevices around the building and in walls, proofing of a few openings of the basement (their likely entry point) is sufficient to yield desired results, rather than closing all points. Besides, the materials used for proofing should be of sound quality and specific standard (which resist rodent gnawing) viz., perforated metal (24 g or higher), expanded metal (24 g), hardware cloth (19 g or higher, with 1.3 cm mesh for rats and 6.3 mm mesh for mice), wire balloons (12 g of galvanized steel or copper wire), cement mortar (1:3), and concrete

Table 2 Physical abilities and characteristic features of commensal rodent pests

| Rodent pests ^a | Physical ability |
|---------------------------|---|
| Norway rat | Excellent burrower—burrows vertically up to 1.5 m depth, extends horizontally to 2–3 m; very good swimmer (1 km) and diver; jumps vertically up to 35 cm, lands safely on its four feet when dropped from a height of 10–15 m; destructive gnawer; enters through openings (1.25–1.5 cm), tunnels, burrows made underneath |
| Roof rat | Excellent and skillful climber, hence found in multistoried buildings and coconut trees; harbors in roofs and upper portions of buildings; climbs wires, cables, pipes, vines, bricks, and rough surfaced walls; good runner, swimmer, and surface burrower; jumps vertically up to 30 cm, passes through openings of 1.25 cm |
| House mouse | Good climber, shallow burrower (10–15 cm depth), good swimmer, destructive gnawer, fast runner; requires very small openings (0.5–0.75 cm) to enter; timid and less neophobic |
| Lesser bandicoot | Extremely good burrower, burrows vertically up to 1–1.5 m depth, extends horizontally up to 5–15 m with several tunnels and plugs to prevent predator entry; excellent swimmer (1–2 km), good climber, and powerful gnawer |
| Larger bandicoot | Predominantly a burrower; burrows vertically up to 1–2 m depth, extends horizontally up to 10–20 m with several tunnels; poor climber, good swimmer, and gnawer; highly aggressive, shows very strong new object reaction; usually enters through tunneling or burrowing; lives outdoors, in warehouses, and in godowns |

^aMost of them are small in size, except larger bandicoot (maximum adult weight of Norway rats—480 g, roof rats—360 g, house mice—35 g, lesser bandicoot—350 g, and larger bandicoot—1600 g), nocturnal in habit, omnivorous in nature, highly adaptable to different kinds of food and environment, with high fecundity rate, and, above all, intelligent and sensitive animals.

(1:2:4). Construction of L-shaped vertical curtain walls (extending 60 cm below ground level, with a horizontal arm that is 30 cm wide at the bottom) around the building, or, as an alternative, construction of the floor 45 cm or more above the ground and made of concrete may be carried out. Extending the foundations below the ground level to 600–900 mm or more prevents burrowing or tunneling from underneath.^[8] Construction of grain storage warehouse with a high plinth (1 m) from ground level without steps but with a ramp parallel to the building by a gap of 1 m, which can be bridged during loading and unloading

operations (Fig. 1), or construction of a 30-cm apron projecting straight or with a slope on top to prevent water stagnation (Fig. 2) at plinth level is a widely used rodent-proofing measure in India. From a long-term point of view, construction of rat-proof warehouses and buildings is the most inexpensive type of control measure when compared to rat proofing later.^[8,9]

FUTURE PROSPECTS

With changes in the human ecosystem caused by industrial, cultural, scientific, technological, and related

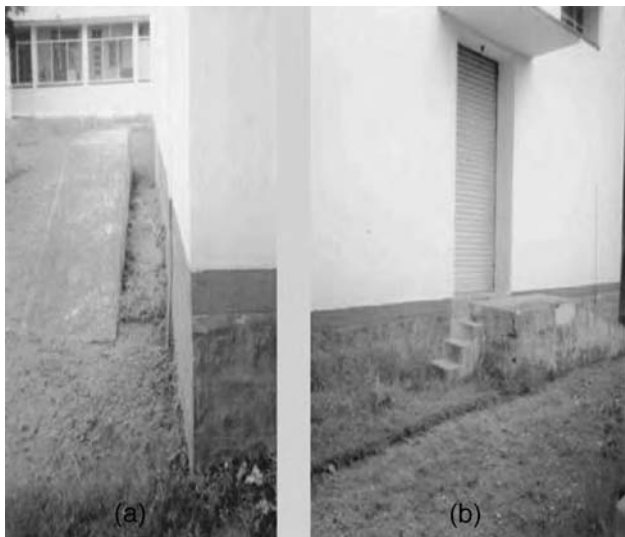


Fig. 1 High plinth from ground level.

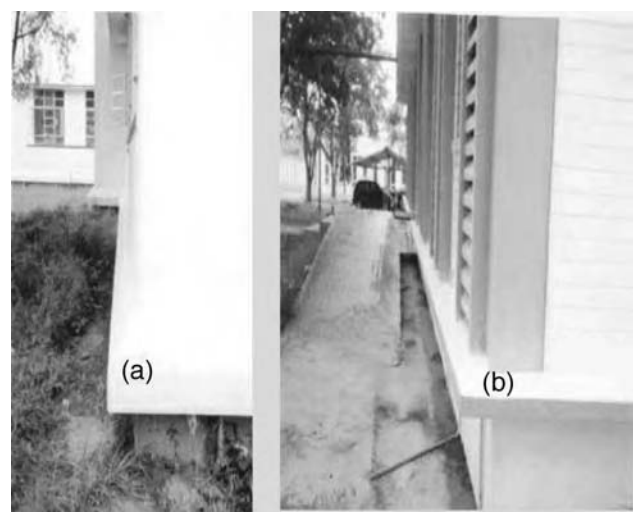


Fig. 2 Projecting aprons.

Table 3 Rodent-Proof grain storage structures for rural and urban areas

| Storage structures ^a | Capacity (tons) | Recommendations |
|---|-----------------|--|
| <i>Underground</i> | | |
| Pits lined with cement bitumen and coat | 0.5–1.0 | Rural, limited application in areas with low water table |
| <i>Above ground</i> | | |
| Metal dehydro bins | 0.5–1.0 | Rural |
| Ferrocement (prefabricated structure) | 10–25 | Rural (can be installed at site) |
| Plywood (with rodent-proof skirting) | 1.0 | Rural and urban |
| Clay (lined with polythene and coated with bitumen) | Up to 5.0 | Rural and urban |
| High-molecular high-density polythene (HMHDPE) bin | 0.5–2.0 up to 5 | Only indoor purpose, easily transportable |
| Warehouse with inverted T-beam | >500 | For bag storage, urban |

^aAll storage structures are fumigable, insect-proof, and moisture-proof.

activities, the types and magnitude of pest problem also have varied. The era of controlling these intelligent pests by lethal approaches has proved insufficient as evidenced by the development of resistance to (both first generation and second generation) anticoagulants in most rodent species. Hence, there is an urgent need to find better management practices that are effective, eco-friendly, and sustainable. Although rodent depredation is of major concern all over the world because of its severity, special attention is required in developing countries. In this context, improving existing storage structures; and designing new structures of low cost, which are moisture-proof, rodent-proof, and termite-proof, with low thermal conductivity and air tightness constructed from reasonably inexpensive materials^[10] suitable for rural and urban areas (Table 3) will play a significant role in solving many developing nations' food problems associated with rodent depredation.

The earlier concept of "rodent exclusion" as a method with high-cost investment and non-availability of appropriate proofing techniques has been revised. Currently, the increased environmental concern—in addition to long-term benefits derived from the application of non-chemical measures and the availability of several standard ready-to-use rodent-proof materials (in developed countries) such as kick plates, baffles, guards, brush strips, thixotropic mouse-proof pastes, composite drains, and fiber glass structures—has made the exclusion measure easier, affordable, and wider in application and acceptance.

CONCLUSION

To achieve long-lasting relief from pest problems, there is a need to integrate rodent exclusion measures with poisoning, trapping, or burrow fumigation. Unless measures to alter the habitation, which potentially

harbors the rodent population, are adopted, there is no use in applying lethal measures that only reduce the number. Resurgence of populations to the original level occurs quickly through survivors and migrants. Therefore control should be viewed in the context of applied ecology and achievement of successful control wherein the population levels have to be kept low, which is possible only by extending proofing and sanitation methods. Hence, there is a greater need now than ever before for evolving suitable strategies based on both chemical as well as non-chemical approaches to achieve the desired results.

ACKNOWLEDGMENTS

The author thanks Dr. Marsh (RE, Specialist in Vertebrate Ecology, Emeritus, Wildlife, Fish, and Conservation Biology, University of California, Davis) for providing the necessary reprints and publications on the subject.

REFERENCES

1. Corbet, G.B.; Hill, J.E. *A World List of Mammalian Species*; Natural History Museum Publications and Oxford University Press: London, UK, 1991; 243 pp.
2. Brookes, J.E.; Rowe, F.P. *Commensal Rodent Control*; Report No. WHO/VBC/87.949; World Health Organization: Geneva, Switzerland, 1987; 1–107.
3. Childs, J.E.; Mills, J.N.; Glass, G.E. Rat borne haemorrhagic fever viruses, special risk for mammalogists?. *J. Mammal.* **1995**, *76*, 664–680.
4. McKee, K.T.; LeDuk, J.W.; Peters, C.J. Hantaviruses. In *Textbook of Human Virology*; Belshe, R.B., Ed.; Mosby Year Book: St. Louis, MO, 1991; 615–632.
5. Hopf, H.S.; Morley, G.E.J.; Humphries, J.R.O. *Rodent Damage to Growing Crops and to Farm and Villages Storage in Tropical and Subtropical Regions*; Centre

- for Overseas Pest Research and Tropical Products Institute: London, England, 1976; 1–115.
6. Jenson, A.G. *Proofing of Buildings Against Rats, Mice and Other Pests*; Ministry of Agriculture, Fisheries and Food, Her Majesty's Stationary Office: London, 1979; 1–31.
 7. Muktha Bai, K. Commensal rodents as pests and methods for their control. *J. Food Sci. Technol.* **1996**, *33* (3), 175–196.
 8. Frantz, S.C.; Davis, D.E. Bionomics and Integrated Pest Management of Commensal Rodents. In *Ecology and Management of Food Industry Pests*; Gorham, J.K., Ed.; FDA Technical Bulletin 4; Association of Official Analysis Chemists: Arlington, USA, 1991; 243–313.
 9. Spencer, D.A. *Storage of Cereal Grains and Their Products*; Anderson, J.A., Alcock, A.W., Eds.; Monograph Series; American Association of Cereal Chemists: St. Paul, MN, 1953; Vol. 2.
 10. Krishnamurthy, T.S.; Shankaramurthy, K.N.; Krishnakumari, M.K. *Grain Storage Structure for Rural and Urban Areas*; Regional Extension Service Centre (Rice Milling), Ministry of Food Processing Industries, Discipline of Infestation Control and

Protectants and Discipline of Cereal Science Technology, Central Food Technological Research Institute, Government of India: Mysore, India, 1992; 1–46.

BIBLIOGRAPHY

- Scott, H.G.; Borom, M.R. *Rodent-Borne Disease Control. Through Rodent Stoppage*; US Department of Health, Education and Welfare, Public Health Service; Center for Disease Control: Atlanta, 1976; 1–33.
- Majumder, S.K. *Role of Rodent Proofing and Sanitation on Rodent Population*, Proceedings of the International Symposium on Bionomics and Control of Rodents, Kanpur, India, September 29–October 2, 1968; Perti, S.L., Wal, Y.C., Srivastava, C.P., Eds.; Science and Technology: Kanpur, 1969, 87–90.
- Howard, W.E.; Marsh, R.E. *The Rat: Its Biology and Control*; US Department of Agriculture: Berkeley, CA, 1981; 1–30. Division of Agricultural Science, University of California, Leaflet 2896.

Rodenticides

Alan MacNicoll

Department for Environment, Food and Rural Affairs, Central Science Laboratories, York, U.K.

INTRODUCTION

Rodents are ubiquitous pests of temperate and tropical climates, and are controlled for crop protection, public health, and conservation reasons. They can cause damage to field crops, forestry, and stored commodities, which may result in serious economic losses and reduce the amount of food available for human consumption in developing countries. Rodents are also carriers of several diseases that can affect both people and livestock. One of the most well known and feared of these diseases is the plague, known as the black death in medieval Europe and still endemic on all continents except Australasia.^[1] Non-indigenous rodents can also have detrimental effects on conservation of rare or declining species, particularly birds, e.g., by consumption of eggs or nestlings. There have been several high-profile programs to eradicate rats on islands to protect endangered wildlife, notably in New Zealand, which has a high prevalence of indigenous flightless ground-nesting birds. Thus there are several reasons why rodents should be controlled or, in some cases, eradicated.

Rodenticides are usually non-specific small molecules (both inorganic and organic) that are self-administered when the rodent eats a bait. Others are applied as fumigant gases or in the form of a “tracking powder” that is ingested during grooming. The lack of specificity of all currently available rodenticides may lead to death of non-target animals that are exposed to the rodenticide, including scavengers and predators that consume dead or dying rodents (secondary poisoning).

FAST-ACTING (OR ACUTE) RODENTICIDES

Fast-acting rodenticides usually cause the death of a rodent within a few hours of ingesting a single, toxic dose of the poison. The innate problem preventing the effective control of some rodent species with this class of rodenticides is the development of conditioned taste aversion (CTA) by individuals that consume a sublethal dose of the poison. Many rodents (e.g., rats and mice) have evolved a feeding strategy of sampling small quantities of novel food and waiting for several hours before returning. If during this postsampling period they experience any ill effects that may have

been caused by eating the novel food, they will not return to that food, nor will they eat any other food that tastes the same. Rodents that have been conditioned in this manner are described as “bait-shy,” and will not consume a lethal amount of bait. Pest controllers have used “pre-baiting” as a technique to overcome CTA. They place a non-toxic foodstuff in places where the rodents will feed and replenish the food frequently over several weeks (which can be very costly in terms of staff time) until the rodents are accustomed to eating this novel source of food. The non-toxic food is then replaced by a rodenticidal bait based on the same food. Animals that have become accustomed to eating the non-toxic food should consume a lethal dose of rodenticide, but even under very favorable circumstances, the most experienced pest controllers would not expect to achieve 100% control. Some remaining rodents may have consumed a sublethal dose and be bait-shy and, for this reason, it is not advisable to use fast-acting rodenticides more than twice a year. In addition, non-target species can also become accustomed to eating the non-toxic food and may be killed when the rodenticide bait is substituted for the prebait.

Sodium Fluoroacetate (1080) and Fluoroacetamide (1081)

Both of these fast-acting rodenticides have similar chemical structures and, ultimately, the mode of action is the same. Fluoroacetamide is converted, in the rodent’s body, to fluoroacetate by enzymic or hydrolytic action. Fluoroacetate is subsequently converted, by a process of “lethal synthesis,” to fluorocitrate which disrupts metabolism in the tricarboxylic acid (Krebs) cycle, a major pathway for producing energy. The processes required to activate both of these rodenticides delay the onset of symptoms for several hours, but not sufficiently to prevent CTA.

Strychnine

Strychnine is an alkaloid isolated obtained from the seeds of *Strychnos nuxvomica*, and has been widely used for medicinal purposes. Both the free alkaloid and its water-soluble salts have been used as rodenticides

since 1640. Strychnine causes severe convulsions prior to death, usually from respiratory failure.

Zinc Phosphide

Zinc phosphide is an inorganic chemical that is widely used as a rodenticide. Acidic conditions in the rodent's stomach releases the gas phosphine, which phosphorylates proteins. Death, after a few hours, usually results from phosphorylation of, and damage to, mitochondrial enzymes involved in respiration and the transport of electrons to oxygen. Some producers market non-toxic pellets to use as a prebait for pellets containing zinc phosphide.

SLOW-ACTING (CHRONIC) RODENTICIDES

Anticoagulants

The Pesticide Manual^[2] lists 19 active ingredients which are currently used as rodenticides and 10 of these are anticoagulants. They are divided into two chemical groups, the 4-hydroxycoumarins and the indandiones, but they all act by inhibiting the vitamin K cycle and by preventing the synthesis of functional blood-clotting proteins. Proteins that are already circulating in the blood degrade over 48–72 hr, and after 4 or 5 days, the rodent dies, usually following a massive hemorrhage. It is this delayed death, and the consequent absence of CTA, that has made the anticoagulants the rodenticide of choice around the world over the last 50 years. In addition, accidental exposure to anticoagulant rodenticides can be detected by a simple blood-clotting test and vitamin K₁ administered as an effective antidote. First-generation anticoagulant rodenticides, typified by warfarin, have relatively low potency and it is usually necessary for rodents to consume bait repeatedly over several days. Second-generation compounds (e.g., difethialone and flocoumafen) are more potent and some products claim that a lethal quantity of anticoagulant may be consumed in a single feed. There is concern in many countries that, at least some of the second-generation anticoagulants may present a hazard, because of their greater potency, to predators and scavengers that eat rodents.

There is considerable debate about the classification of anticoagulant rodenticides into the first and second generations because of different rankings of potency in different species. The following is a list from lowest to highest potency, based on oral LD₅₀ values^[2] for *Rattus norvegicus*: pindone; warfarin; coumatetralyl; chlorophacinone; diphacinone; difenacoum; bromadiolone; difethialone; brodifacoum; flocoumafen.

Resistance to the first generation of anticoagulant rodenticides was discovered in rats (*R. norvegicus* and *R. rattus*) and mice (*Mus musculus domesticus*) in the United Kingdom around 1960, approximately 10 years after the first use of anticoagulant rodenticides. Similar resistance has now been reported in many countries across the globe. The second-generation anticoagulant rodenticides were specifically developed to overcome resistance, but resistance to several of these more potent analogs has now been detected.^[3–5] Continued use of anticoagulant rodenticides against these populations of resistant rats is likely to maintain the selection pressure toward higher prevalence and degrees of resistance.

Calciferol (Vitamin D)

Both ergocalciferol (vitamin D₂) and cholecalciferol (vitamin D₃) are used as rodenticides. They are sometimes classed as acute or fast-acting rodenticides, but deaths rarely occur less than 4 days after bait consumption. The mode of action of both calciferols is to mobilize calcium from bones and cause deposition of crystals of calcium salts in soft tissue such as liver and kidney. However, there is evidence that rodents, particularly rats, can detect early symptoms of the effects of calciferol and develop CTA. Prebaiting can be used, as described above, to partially overcome this problem.

Alpha-Chloralose

Alpha-chloralose is an organic narcotic that has been used for many years as a seed dressing to repel birds. It is also used as a rodenticide and is particularly useful against small rodents such as mice because it acts by slowing metabolism and reducing body temperature.

Bromethalin

Bromethalin was identified as a potential rodenticide in the late 1970s. Its mode of action is to uncouple oxidative phosphorylation in the central nervous system, reducing energy production, and increasing pressure in the cerebrospinal fluid. Death occurs after approximately 2–3 days, and there is no evidence of bait-shyness.

FUMIGANTS

Two fumigant gases are used to control rodents, usually by treatment of burrows that must be located and sealed, but are also used for control of rodents in ships, containers, and warehouses. Both hydrogen cyanide and phosphine can be generated in situ by

the action of water or dilute mineral acids on their respective metal salts. Fumigation can be an effective means of rodenticide application and rodent control, but is limited by the need to locate and gain access to all of the burrows used by rodents, the potential risk to nontarget species including people, and the requirement (in some countries legally enforced) for application by trained fumigation technicians.

BIOLOGICAL RODENTICIDES

Pathogenic microorganisms have been used to control rodents, in the same way that the myxoma virus has been used to control rabbits. Cultures of *Salmonella enteritidis* were extensively used for rodent control in Europe in the first part of the twentieth century, but their use was frequently associated with outbreaks of enteritis in people, and they were largely superseded by the much safer anticoagulants. Although the World Health Organization and the Food and Agriculture Organization recommended^[6] in 1967 that *Salmonella* should not be used for rodent control, it is still used in some countries.

ALTERNATIVES TO RODENTICIDES

Many innovative techniques have been used, often in developing countries where rodenticides can be relatively expensive, for killing rodents or preventing damage. These include a whole range of traps and snares, digging out of burrows, driving into nets or other traps, stalking at night by lamplight, electrocution, barriers to protect crops, stores, or trees, barriers with traps at intervals, and scaring devices. In the last 5 years of the twentieth century, the concept of ecologically based management of rodent pests (EBMRP) was developed by a group of prominent rodent ecologists. The concept of EBMRP is an extension of integrated pest management (IPM), and uses the principle that management of rodent populations is best achieved by integrating knowledge about the animal's biology and behavior with a well-organized control scheme,^[7] rather than solely relying on the use of chemical rodenticides.

FUTURE PROSPECTS

Although there are undoubtedly under-exploited opportunities to limit rodent numbers by EBMRP, there will always be circumstances where it is necessary to quickly reduce, or eradicate, rodent infestations. It is likely that one or more of the rodenticides described above will provide an effective means of controlling rodent numbers. However, all of these rodenticides are broad-spectrum and non-specific, which can reduce their usefulness because of their potential hazard to people, livestock, and nontarget wildlife species. Rodent-specific toxins are needed to provide safe, effective, and humane rodent control without the risk of poisoning other species.

REFERENCES

1. Perry, R.D.; Fetherstone, J.D. *Yersinia pestis*—Etiologic agent of plague. *Clin. Microbiol. Rev.* **1997**, *10*, 35–66.
2. Tomlin, C.D.S. *The Pesticide Manual—A World Compendium*; British Crop Protection Council: Farnham, UK, 2000.
3. Kerins, G.M.; Dennis, N.J.; Atterby, H.; Gill, J.E.; MacNicoll, A.D. Distribution of resistance to anticoagulant rodenticides in England 1995–98. In *Advances in Vertebrate Pest Management—Volume 2*; Pelz, H-J., Feare, C., Cowan, D.P., Eds.; Filander Verlag: Furth, Germany, 2001; 149–159.
4. Lodal, J. Distribution and levels of anticoagulant resistance in rats (*Rattus norvegicus*) in Denmark. In *Advances in Vertebrate Pest Management—Volume 2*; Pelz, H-J., Feare, C., Cowan, D.P., Eds.; Filander Verlag: Furth, Germany, 2001; 139–148.
5. Pelz, H-J. Extensive distribution and high frequency of resistance to anticoagulant rodenticides in rat populations from northwestern Germany. In *Advances in Vertebrate Pest Management—Volume 2*; Pelz, H-J., Feare, C., Cowan, D.P., Eds.; Filander Verlag: Furth, Germany, 2001; 161–170.
6. Anon. Joint FAO/WHO Expert Committee on Zoonoses. 3rd Report. In *WHO Technical Report Series No. 378*; WHO: Geneva, Switzerland, 1967.
7. Singleton, G.; Hinds, L.; Leirs, H.; Zhang, Z. *Ecologically Based Rodent Management*; Australian Centre for International Agricultural Research: Canberra, Australia, 1999.

Rotterdam Convention and Pesticides

Barbara Dinham

Eurolink Centre, Pesticide Action Network UK, London, U.K.

INTRODUCTION

When chemical pesticides were introduced 50 years ago, little attention was paid to the environmental and health impacts. With the rapid expansion of use in the 1950s, understanding gradually increased of the consequences of exposure to certain chemicals. Wide-ranging impacts began to be identified, including: environmental persistence and effects on birds and wildlife; residues in soil, water, and air; residues in food; human poisonings from acutely toxic pesticides or long-term health impacts such as cancer; and pest resistance, often leading to dramatic crop losses.

With almost 1000 different pesticides and thousands of formulations on the market to control insects, diseases, weeds, and other pests, action was clearly needed to protect human health and the environment. International standards recommended that governments establish a registration system to authorize each formulation of a pesticide for each specific crop or other use. Concern with some pesticides led governments to ban or restrict them to a limited number of uses. Few developing countries can fully implement a registration scheme, and they are often unaware of bans imposed elsewhere. Recognizing these problems, in the early 1980s, governments, international organizations, and public interest groups began to demand action to provide a warning system to help developing countries regulate or ban the use of hazardous pesticides.

The Rotterdam Convention on Prior Informed Consent Procedure for Certain Hazardous Chemicals and Pesticides in International Trade^[1] is the outcome of 15 years of activity on trade in hazardous chemicals. Adopted on 10 September 1998 in Rotterdam, the Netherlands, the Convention was signed by 73 countries^[2] and by June 2001 had been ratified by 14 parties. It will become legally binding after 50 countries have ratified.

The Convention takes an important step toward protecting humans and the environment from highly toxic chemicals. For the first time, it will help monitor and control trade in dangerous substances, circulate better information about health and environmental problems of chemicals, and prevent unwanted imports of certain hazardous chemicals.

Central to the Rotterdam Convention is the system of Prior Informed Consent (PIC), a means of obtaining and disseminating decisions of importing countries about their willingness to receive shipments of certain

chemicals, and ensuring compliance to these decisions by the exporter. To be included in PIC, a pesticide must be banned or severely restricted for health or environmental reasons by two countries in two different regions of the world—indicating that its adverse effects are a “global concern.”

But focusing on banned or severely restricted pesticides may only touch the tip of the iceberg. Industrialized countries rely on trained and informed users able to apply good practice as safeguards: in developing countries where pesticides are often used under conditions of poverty, these measures cannot be applied. Furthermore, older—and often more hazardous—pesticides are often cheaper, making them attractive to poorer farmers. The Convention recognizes that “severely hazardous pesticide formulations” should be included in PIC if they cause health or environmental problems in developing countries or in Eastern Europe—termed “countries with economies in transition”—in the Convention.

THE HISTORY OF PIC

A PIC system was first proposed in the early 1980s as part of the International Code of Conduct on the Distribution and Use of Pesticides, negotiated by governments in the Food and Agriculture Organization (FAO) of the UN. Some governments resisted the concept, and the Code was adopted in 1985 without any reference to PIC. But intense pressure from non-governmental organizations (NGOs) and others won support, and the principle was accepted in 1987. It took until 1989 to establish the wording and issue a revised version of the Code.^[3] That same year, the UN Environment Programme (UNEP) included an identical provision in the London Guidelines on the Exchange of Information on Chemicals in International Trade, and a voluntary system was put in place with the FAO acting as the Secretariat for pesticides and UNEP for industrial chemicals. The first pesticides were added in 1991, and by 1995, 22 pesticides and five industrial chemicals were included.

From Voluntary to Legally Binding

The issue of transforming the voluntary scheme into a legally binding international Convention was first

mooted in 1992 at the United Nations Conference on Environment and Development (UNCED).^[4] In November 1994, the FAO Council meeting agreed to proceed, and this was followed in May 1995 by a decision of the UNEP Governing Council. The two organizations convened an Intergovernmental Negotiating Committee (INC) to draft and agree international legally binding instrument.

Banning Exports of Banned Pesticides

An alternative to PIC strongly advocated at the time was to stop all exports of banned pesticides. However, unless action to limit the market for a banned pesticide could be taken, banning exports could encourage companies to relocate production, possibly in a country with less stringent controls. Preventing the export of banned pesticides would have no effect on severely restricted chemicals. Without a PIC system, a developing country could unwittingly allow the import of banned or severely restricted pesticides, ignorant of action taken by some governments. Many developing countries maintained that an export ban could limit their development, as alternatives were more expensive, and that import decisions should rest with them. PIC does not prevent individual countries from deciding that their banned pesticides should not be exported, but does ensure that regulatory actions are widely shared.

HOW THE CONVENTION IS OPERATED

In negotiating the text of the Rotterdam Convention, governments built on the experience gained in the voluntary PIC. As a mark of its importance, the Convention began immediately on a voluntary basis, with FAO and UNEP continuing as an interim Joint Secretariat.

Designated National Authorities

To participate in PIC, governments must appoint a Designated National Authority (DNA). By December 2000, 170 governments had appointed a DNA or a focal point. When ratifying the Convention, DNAs must be authorized to carry out administrative functions such as receiving, transmitting, and circulating information.

Notifying Regulatory Actions

When a government bans or severely restricts a pesticide, it must notify the Joint Secretariat within 90 days. Governments need to demonstrate that their action is final and that it was based on a risk evaluation,

including a review of scientific data, and the Secretariat will validate the notification. Once two valid notifications from different PIC regions have been received for the same pesticide, it becomes a candidate for PIC.

The Chemical Review Committee

The Convention set up a Chemical Review Committee to consider notifications, and advise the Conference of the Parties (CoP—this will replace the INC after ratification). A parallel structure operates in the voluntary phase, with an Interim Chemical Review Committee (ICRC). The Committee will review PIC notifications, and—when they meet the agreed criteria—draft a Decision Guidance Document (DGD).

Two Routes to be “PIC-ed”

Pesticides in the voluntary PIC were carried forward, and new pesticides continue to be added. By June 2001, the process included 26 pesticides and five industrial chemicals (Table 1).

There are two routes for adding pesticides to the Convention. Under Article 5, a ban or severe restriction in any two regions triggers PIC if the action is taken for health or environmental reasons. Governments have decided that the PIC regions would be: Africa (48 countries), Latin America, and the Caribbean (33 countries), Asia (23 countries), Near East (22 countries), Europe (49 countries), North America (2 countries: Canada and US), Southwest Pacific (16 countries).

The second route is covered in Article 6, and addresses “severely hazardous pesticide formulations.” This category applies only to pesticide formulations found to be causing health or environmental problems under conditions of use in developing countries, or countries with economies in transition. These pesticides may not have been banned, but—generally because of high toxicity—cause poisonings and deaths when used without extreme caution. Governments must submit evidence based on a “clear description of incidents related to the problem, including the adverse effects and the way in which the formulation was used.” Nevertheless, this kind of evidence is rare, and collecting information is difficult: incidents take place far from medical facilities; many farmers are unaware of the active ingredients of pesticides they use; and it is common to use mixtures of several pesticides. The ICRC is investigating how to deal with these problems.

Import Decisions, Information, and Website

Once a pesticide is included in PIC, the DGD is circulated to all governments who must decide whether to

Table 1 Pesticides covered by the interim PIC procedure, November 2000

Banned or severely restricted pesticides^a

2,4,5-T (dioxin contamination)

Aldrin

Binapacryl (INC6)^a

Captafol

Chlordane

Chlordimeform

Chlorobenzilate

DDT

Dieldrin

Dinoseb and dinoseb salts

1,2-Dibromoethane (EDB, or ethylene dibromide)

Ethylene dichloride (INC7)^a

Ethylene oxide (INC7)^a

Fluoroacetamide

HCH, mixed isomers

Heptachlor

Hexachlorobenzene

Lindane

Mercury compounds

 mercuric oxide

 mercurous chloride, Calomel

 other inorganic mercury compounds

 alkyl mercury compounds

 alkoxyalkyl/aryl mercury compounds

Pentachlorophenol

Toxaphene (INC6)^a

Severely hazardous pesticide formulations^b

Monocrotophos

Methamidophos

Phosphamidon

Methyl parathion

Parathion

^aIndicates that these four pesticides were added to the PIC list at the 6th and 7th International Negotiating Committee meetings.

^bOnly certain formulations of these severely hazardous pesticides are included.

(From Ref.^[5].)

consent to or prohibit its import. Import decisions are posted on the PIC website, and circulated biannually. Governments in exporting countries must ensure that their exporters comply. Of course, many countries are both importers and exporters and under the rules of international trade, a country cannot ban the import of a pesticide that is manufactured and used nationally.

An important tool is the PIC Circular, updated every six months by the Secretariat. Circulated in hard copy and on the website,^[5] it includes new bans and severe restrictions, importing country responses, and general progress reports. For the first time, it is easy to access sound information on government regulatory actions, even if these do not meet all the full PIC criteria.

The Convention—More Than PIC

Information exchange is an important principle promoted under Article 14 of the Convention. Developing countries lack resources to undertake extensive evaluations of pesticides and governments are encouraged to share scientific, technical, economic, and legal information on chemicals within the scope of the Convention, as well as other information on their regulatory actions.

BUILDING CAPACITY/IMPROVING REGULATIONS

The process of identifying problem pesticides through PIC will be slow, and there are limitations. In some cases, for example, governments will have no easy substitute, although this may increase the incentive to seek safer and more appropriate alternatives, including Integrated Pest Management strategies.

Financial resources are needed, not only to allow the Secretariat to meet its obligations, but also to ensure that regulators in developing countries can participate in workshops and training sessions. In poorer countries, with competing demands on scarce resources, chemical regulation is not always a priority. The status of an international Convention gives PIC the attention it requires to be effective, and should help attract the necessary funds.

PIC is just one tool, although an important one, in the regulation of pesticides. With good training and additional resources, PIC can play a central role as part of capacity-building initiatives to help governments improve their ability to regulate pesticides, and to look for products and strategies that reduce the dependence on hazardous chemicals.

REFERENCES

1. Rotterdam Convention on the Prior Informed Consent Procedure for Certain Hazardous Chemicals and Pesticides in International Trade, UNEP and FAO, Text and Annexes, **January 1999**.
2. The signatory countries can be found on the PIC website: www.pic.int/. The Convention closed for signatures in September 1999: countries which have not signed accede to, rather than ratify, the Convention, to the same effect.
3. *International Code of Conduct on the Distribution and Use of Pesticides (Amended Version)*; FAO, 1989. The Code is currently being revised and updated.
4. United Nations Conference on Environment and Development, Agenda 21, Chapter 19, Environmentally Sound Management of the Toxic Chemicals, Including Prevention of International Illegal Traffic in Toxic and Dangerous Products, UNEP, Nairobi, **1992**.
5. Convention text and PIC website (www.pic.int/).

Rouging

Hector Achicanoy

*Departamento de Agronomía, Universidad Nacional Colombia Sede Medellín,
Medellín, Colombia*

INTRODUCTION

The rouging of seemingly infectious plants can be a successful measure in reducing the level of inoculum or even destroying the pathogen altogether. Rouging is particularly effective against viral diseases and some bacterial diseases. However, host elimination in fungal diseases is generally not as successful. Sometimes, the elimination of voluntary plants, overgrowths, and alternating hosts is enough to reduce or destroy the inoculum.

ROUGING

When one tries to develop and apply new methods of control of plant diseases, the objective should be a rational, effective, and sure control at a minimum cost. You can only achieve the control of these diseases by means of a procedure, but in most cases, it demands the use of multiple measures and implies an integrated program of manipulation of the atmosphere and control methods. Generally, the decision on the control of diseases is based on the theory of the economic threshold, which refers to the lowest population level that is able to cause economic damage or reduce in production. However, it should be kept in mind that the decisions based on thresholds are uncertain and they only have validity for certain crops in a stage of growth of the cultivation and under certain environmental conditions.^[1]

Therefore, the measures for the control of diseases should be preventive, and if it is possible, based in a prediction system that allows to determine the probability when a plant disease reaches the threshold of economic damage.^[2]

It is known that all infectious diseases have three epidemic parameters that govern its advancement or development: quantity of initial inoculum; infection rate; and time. This means that the methods of control of diseases should have an effect on those parameters, particularly the initial inoculum and the infection rate.^[2] The cultural methods are based on the farmer's activities or cultivation dedicated to avoid plant pathogens (when the pathogen is introduced by means of ineffective quarantines) or to reduce plant pathogens in the plantation (when the pathogen is able to settle down).

The cultural methods include the elimination of the plant pathogen and the application of certain favorable conditions for the host and unfavorable conditions for the pathogen. In the elimination of the pathogen, some biological, physical, and chemical methods were also used.^[1]

The elimination of the plant pathogen take in strategies dedicated to the destruction or reduction of the inoculum after it has been able to settle down in the crop host and has been generally carried out by means of cultural measures as systematic elimination of plants diseases, crops rotation, and sanitary methods.^[1]

In certain cases, particularly in the viral diseases and some bacterial diseases, the rouging of the seemingly infectious plants is a successful measure to destroy the pathogens or to reduce considerably the quantity of inoculum. In the plants diseases caused by fungi, the elimination of the host is not generally successful. Sometimes, the elimination of voluntary plants, overgrowths, and alternating hosts is enough to destroy or reduce the inoculum.^[1]

SUCCESSFUL APPLICATIONS OF THE ROUGING

In general, the elimination of the infected plants is a recommended measure when the dissemination of the infection is from plant to plant, keeping in mind that the infection is not very advanced. It is mainly used in seedbeds, greenhouses, nurseries, orchards of citric, perennial crops, and in the production of propagation material free of plant diseases.^[1]

In some tropical countries, as Colombia, the rouging has been successful in plant diseases like the red ring of the coconut tree caused by the nematode *Radi-naphelenchus cocophilus*, the moko of the banana or plantain caused by the bacteria *Pseudomonas solanacearum*, and the starry wound of the cocoa originated by the fungus *Rosellinia pepo*. In certain viral diseases, as the papaya ring spot (PRV), in combination with other cultural measures, the elimination of infected plants has been able to reduce the quantity of inoculum.^[3]

However, the campaigns to avoid the dispersion of some fungal diseases, as the black sigatoka in banana and plantain caused by *Mycosphaerella fijiensis* and

the rust of coffee caused by *Hemileia vastatrix*, were a failure due particularly to the biology of the pathogens related to their discharges, reproduction rates, and infection; to the condition of pathogen polycyclics and their disease gradients; and to the primary and secondary dispersions, mainly through the air movement of the inoculum.

In the case of the black sigatoka, the factor of importance in the failure of the elimination of plant disease was the very advanced state of the infection. The result was the presence of several sources of secondary inoculum with a gradient of the uniform disease due to the effect of multiple infections by means of aloinfection or exodemy and autoinfection or esodemy.^[4]

The above mentioned means that the measure of elimination of the host is only successful for those plant diseases where the inoculum has a rate of low reproduction; the dispersion form is not by means of the wind, and in some cases not by rain or air vectors, especially when the infection is in its initial stage.^[1,5] The measure of rouging is recommended for certain plant diseases like the roots wilt, the vascular diseases, some plants diseases caused particularly by virus, diseases that are transmitted in form mechanics or for terrestrial vectors, and some bacterial diseases with origin in the soil or of mechanical transmission.^[1]

COSTS AND BENEFITS OF ROUGING

The economic importance of the plants diseases should not only be measured by the true damage that they cause, but also by the costs in the measures of prevention and control and the limitations that some cultivations or varieties show in certain agricultural areas. The systematic elimination of the plant diseases implies an additional cost particularly in the agricultural production in perennial crops due to the necessity of using manpower in the destruction of the affected

plants; however, this invested cost is minimized, and is considered a benefit when avoiding a possible epidemic which is characteristically devastating that can mean the ruin the crops of the farmer and his neighboring producers.^[1]

At the present time, world agriculture in spite of technological advances, demands more and more and better handling practices for the control of plant diseases that cause serious economic losses. However, the use of different methods to avoid losses in the crops and to improve the quality of their products should be economically feasible and environmentally acceptable. This means that continuous development and successful implementation of strategies of control of plant diseases need detail understanding of all the factors that intervene in the development of the infectious plants.^[1,2]

REFERENCES

1. Achicanoy, H. *Introduction to Plant Pathology*, 1st Ed.; Center of Publications National University of Colombia: Medellin, 2001; 320 pp, In impression.
2. Achicanoy, H. *Principles of Plant Disease Epidemiology*, 1st Ed.; Center of Publications National University of Colombia: Medellin, 2000; 189 pp.
3. Achicanoy, H. Integrated management of the papaya ring spot virus. *Fitopatología Colombiana* **1995**, *19* (1), 67-71.
4. Robinson, R. *Plant Pathosystems*; Springer-Verlag: Berlín, 1976; 184 pp.
5. Zadoks, J.; Schein, R. *Epidemiology and Plant Disease Management*; Oxford Press: London, 1979; 428 pp.

BIBLIOGRAPHY

- Palti, J. *Cultural Practices and Infectious Crop Diseases*; Springer-Verlag: New York, 1981; 98-118.

Runoff

Lívía Nemeth Konda

Analytical Chemistry Department, Institute for Veterinary Medicinal Products,
Budapest, Hungary

INTRODUCTION

Urban, industrial, and agricultural activities produce increasing amounts of potential pollutants, which are introduced to cropped and uncropped areas. The present use of chemicals, including pesticides in agriculture, is an extensive anthropogenic source for the release of xenobiotics into the environment. The greatest potential for unintended effects of pesticides is through the contamination of the soil and the hydrologic system. The fate and the behavior of pesticides in the environment involve several different, and often simultaneous, phenomena—including chemical, biological, and photochemical degradation; transport; and accumulation. The transport of pesticides can take place through runoff, erosion, leaching, volatilization, and wind erosion. Surface runoff is one of the most significant sources of pesticides in surface water.

Runoff constitutes parts of precipitation, snow melt, or irrigation water that appears in uncontrolled surface streams, rivers, drains, or sewers. Runoff may be classified according to the speed of appearance after rainfall or melting snow as direct runoff or base runoff, and according to the source as surface runoff, storm interflow, or groundwater runoff.

Environmental impact, theory, factors affecting pesticide runoff, testing methods, relevant management practice, and risk assessment constitute the topics that will be discussed with regard to runoff processes to decrease the loss of pesticides and to prevent the pollution of the environment.

IMPORTANCE OF RUNOFF AND ITS IMPACT ON ENVIRONMENT

The widespread use of organic pesticides over the past half-century has led to the contamination of the environment, especially the hydrologic system of the world. A portion of herbicides applied to forests, croplands, roadsides, and gardens is inevitably lost to water bodies, either directly (through runoff) or indirectly (by leaching through groundwater into ephemeral streams and lakes). Water monitoring in many countries has revealed contamination by various toxic chemicals, particularly pesticides, nutrients (nitrogen and

phosphorus), and sediment. The amount of pesticide leached below the root zone by worst-case rainfall events can reach up to 5% of the applied mass, depending on the soil and pesticide properties. When there is no heavy rainfall following the application of chemical, the mass annually leached below the root zone is in the range of 0.1–1%. The mass lost by leaching seems to be generally smaller than the amount lost by runoff, depending on the slope of the fields. Single rainfall events can cause substantial losses to surface waters (Table 1). Episodic pollution events, such as runoff, can lead to a short-term contamination of aquatic ecosystem with pesticides. It has been estimated for a wide range of pesticides that 1–2% of the applied mass can be lost in a single runoff event.^[1] It has been calculated that the Mississippi River carries an annual mass of 160 t of atrazine, 71 t of simazine, 56 t of metolachlor, 18 t of alachlor, and 3.5 t of acetochlor into the Gulf of Mexico.^[2] The contamination of several European rivers by pesticides, e.g., Arno (Italy), Elbe (Germany), and in Greek and Swedish natural surface waters, has been detected as well. Pesticides frequently occur in the streams and lakes throughout the winter, which illustrates that a short-term exposure caused during spraying season following runoff events results in a long-term contamination of surface waters.

The presence of pesticides in groundwater and surface water may constitute considerable negative effects to human health and ecosystems. Pesticide residues in drinking water may affect human health, while ecosystems may be affected by a loss of biodiversity and a decrease in the population of sensitive living systems. According to the survey of the Environmental Protection Agency (1990, 1992), a considerable number of wells contained pesticide at trace levels. Pesticides were found in 10.4% of community water system, and in 4.2% of rural domestic wells. Nevertheless, less than 1% of all the wells surveyed had pesticide concentration slightly above levels considered safe for human health. In the aquatic environment, pesticides may cause stress within aquatic communities and may radically alter community structure as a whole. The main output of several studies on the impact of different pesticides to aquatic organism is summarized in Table 2. It appears that a single universal maximum limit on the pesticide application in catchments, as suggested by

Table 1 Typical loss of some frequently used pesticides due to runoff on agricultural soil

| Pesticide | Loss in % of total applied amount | Concentration in runoff water (mg/L) | Remark |
|---------------|--|--------------------------------------|---|
| Atrazin | <0.3 | 0.002 | 30% of the amount in runoff is adsorbed on sediment |
| Simazine | 0.3–1.5 | | |
| Simetryn | 5.4–10 | | |
| Prometryn | 8 | | Bare field soil |
| Hexazinone | 0.32 | | |
| Acetochlor | 0.19 | | |
| Metolachlor | 0.04 | 5.1 | Significant amount of hydrophilic sulfonic acid-type metabolites could be formed |
| Diuron | 0.07 33.3 ^a 39.5 ^b | | |
| Isoproturon | 0.3–1.2 | 60 | |
| Glyphosate | 8.5 ^a 12.5 ^b | | |
| Diazinon | 0.13–2.1 | 0.88 | 10% of amount in runoff is adsorbed on sediment; concentration in sediment reached 2 mg/g |
| Chlorpyrifos | 0.08–0.9 | | |
| Fenitrothion | 0–0.08 | 0.62 | 10% of the amount in runoff is adsorbed on sediment; concentration in sediment reached 4 mg/g |
| Pendimethalin | 0.81–1.22 | 18–81 | |
| 2,4-D | 0.2–14 | 45.5 | |
| Dicamba | 0–7.8 | 1.59 | |
| Diclosulam | 0.6–5.2 | | 3% of the amount in runoff is adsorbed on sediment |
| Endosulfan | 2; 1.4 | | Sum of endosulfan and metabolites |
| DDE | 3 | | Bare field soil |
| Trifluralin | 5 | | Bare field soil |
| Thiobencarb | 0.2–0.9 | | |
| Bentazon | 1.4 | | |
| Cyanazine | 2 | | |

Please note that these data should not be applied to any situation because they are greatly influenced by soil type, climate, geographical conditions, cultivation, and pesticide application method.

^aFrom sand.

^bFrom concrete.

many regulatory authorities, does not provide adequate protection for the aquatic environment. Rather, it is advocated that flexible limits on the application of pesticide be developed, in line with the potential risk of contamination to surface water and groundwater, and the fragility of the aquatic environment.

THEORY AND FACTORS AFFECTING RUNOFF

The fate and the environmental behavior of pesticides are influenced by a number of physical, physicochemical,

biochemical, pedological, and climatic factors; crop type; cropping practices; and management methods.

Physicochemical Properties of Pesticide and the Soil

While various physicochemical processes affect the fate of agrochemicals that established contact with soil, the sorption to the solid matrix of soil is one of the most important phenomena that has influences on transportation and transformation processes. Pesticides,

Table 2 Examples of pesticide impact to aquatic environment

| Pesticide | Effectuated ecosystem | Species | Adverse biological effect |
|--|-----------------------|---|---|
| Atrazine | Wetland | African clawed frog | Decreased testosterone levels, demasculinization, hermaphrodites |
| Copper-based pesticides, organophosphate pesticides | Marine | Crab, clam, oyster, shrimp, grass shrimp | Toxicity, accumulation, acetylcholinesterase inhibition, decreased population |
| Chlorpyrifos, carbofuran, diazinon, methyl parathion | River | <i>Ceriodaphnia dubia</i> , neomysis | Acetylcholinesterase inhibition, toxicity |
| Organochlorine pesticides | Estuary, river | Bacteria, protozoa, larvae, amphibian, insect communities, fish | Decreased population accumulation in tissue |

which are strongly adsorbed to soil, are not carried downward through the soil profile with percolating water. However, strongly adsorbed pesticides can be carried with eroded soil particles by surface runoff. Dissolved pesticides or those adsorbed to eroding soil particles can lead to a contamination of surface water resources. This is of particular concern with persistent pesticides on highly erodible soils.^[3] The adsorption rate is affected by the properties of the pesticide and the soil characteristics as well. With respect to mineral components, the content and the nature of organic matter in the soil play a key role in the performance of applied pesticides.^[4] On the other hand, in arid zones and in regions where the organic matter content of soil is low and long period of dryness is present, the mineral surface is the main active site to adsorb pesticides. The four parameters that have been most frequently used for the prediction of the mobility of chemicals in soil environment include: 1) the *n*-octanol/water partition coefficient (K_{ow}); 2) the water solubility of the compound; 3) the adsorption coefficient normalized by the organic carbon content of soil (K_{oc}); and 4) the soil dissipation half-life. K_{ow} is a useful parameter in the prediction of adsorption on soil, which is related to the hydrophobicity of the test substance. Water solubility provides an estimate of the maximum aqueous concentration that is likely to be encountered. Organic chemicals with low aqueous solubility and high *n*-octanol/water partition coefficients are considered to be more strongly adsorbed by soil compared to compounds that are more water-soluble and have a lower K_{ow} . A pesticide's tendency to be adsorbed by soil is expressed by its adsorption coefficient. Adsorption coefficient is defined as the ratio between the concentration of the substance in the soil and the concentration of the substance in the aqueous phase at adsorption equilibrium. The adsorption coefficient normalized to the organic carbon content is a useful indicator of the binding capacity of a chemical on organic matter of the soil, and allows comparison to

be made between different chemicals. The soil dissipation half-life serves as a rough indicator of persistence of a chemical in situ.

Pesticide Formulation

The rate of the movement of pesticide also depends on the type of formulation. The most common types of pesticide formulation are sprayable (e.g., emulsifiable concentrate, wettable powder, suspension concentrate, water-dispersible granules), granular, and controlled-release formulations. Granular formulation may have a greater effect on the transport of a pesticide than sprayable formulation, particularly when rainfall occurs immediately after application. One of the greatest benefits of controlled-release formulation is that the amount of active ingredient applied per area in the field can be reduced (in contrast to other formulation); in this way, environmental contamination may be lessened.

Soil Characteristics

Permeability, soil texture, soil structure, and soil moisture are the main factors that determine the amount of water percolating through the soil profile, and the amount of water running off the surface. Permeability is a measure of how fast water can move vertically through the soil. It is affected by the texture and the structure of the soil. Soil texture describes the relative percentage of sand, silt, and clay content of the soil. Soil structure describes how the soil is aggregated. Uncompacted, coarse-textured soils, such as sandy soils with low water-holding capacity, generally have high potentials for leaching of pesticides to groundwater, but low potentials for surface loss to streams and lakes. Fine-textured soils, such as clay and clay loam, generally have low infiltration capacities; thus

surface runoff is relatively high compared to percolation. As mentioned earlier, organic matter in a soil determines its potential for pesticide adsorption. Moreover, a high content of organic matter may reduce the potential for surface loss by providing good soil aggregation in the plow layer, which increases the infiltration rate and therefore reduces runoff and erosion. Soil moisture is also another major factor that affects runoff. If the soil is already wet or saturated before rainfall or irrigation, excess moisture will lead to runoff.

Climatic Condition

The main meteorological factors affecting runoff include the following: type of precipitation, rainfall intensity, rainfall amount, rainfall duration, distribution of rainfall over the drainage basin, direction of storm movement, antecedent precipitation and resulting soil moisture, and other meteorological and climatic conditions that affect evaporation—such as temperature, wind, relative humidity, and season. The assessment of the potential for pesticide loss from the surface should include an evaluation of the site-specific water balance. Water originating from precipitation or irrigation infiltrates into soil, or runs off the soil surface. The fraction of water that infiltrates compared to the fraction that runs off depends largely on the intensity of the precipitation and the infiltration capacity of the soil. During winter, soil is likely to be frozen and impermeable to water. In springtime, snowmelt, rain, and low evaporation rates generate wet soil conditions. The potential for runoff is high during this period because the near-saturated or partially frozen soil has low water infiltration capacity. In addition, runoff and erosion are often aggravated by the lack of crop canopy, which protects the soil surface from direct raindrop impact. During summer, high rates of evaporation and plant water uptake may reduce soil moisture content. Summer rains only partially recharge the soil profile, and the soil's moisture-holding capacity is typically not exceeded. Except during high-intensity thunderstorms, runoff and erosion potential are generally low during the summer. On the other hand, the seal formation at the soil surface is significantly affected by raindrop kinetic energy, and soil sealing enhances runoff and soil erosion.

Geographical and Hydrological Conditions of the Site

The main geographical and hydrological characteristics affecting runoff are drainage area, basin shape, elevation, slope, topography, direction of orientation, and the presence of ponds, lakes, reservoirs, sinks, etc.

in the basin, which prevent or alter the course of runoff from continuing downstream. The presence of sinkholes, cracked bedrock, or confining layers in the bedrock significantly affects the vertical movement of water. Sinkholes pose a high risk for groundwater contamination by pesticides if runoff from fields where such pesticides are applied can reach sinkholes.

Effects of Cultivation Method

At the agronomic level, farmers may influence the fate of applied pesticides through farming practices. Variations in farming practices contribute to variability in the loads of pesticides by runoff to surface waters. There are many promising possibilities to influence transport processes; however, the inverse relation between leaching and runoff leads to a dilemma when the attempt is made to reduce the overall loss of agrochemicals to surface and subsurface water. Minimizing pesticide movement in surface runoff involves several approaches. It has generally been assumed that conservation tillage practices result in reduced runoff volume, soil erosion, nutrient, and pesticide losses, except in cases where infiltration is limited. On the other hand, the application of pesticides and nutrients on the surface without incorporation may increase potential losses. Contouring, terracing, and strip cropping also have the potential to reduce runoff losses. Grassed waterways and buffer strips retard the transport of sediment and water from the field; in this way, they may reduce the delivery of pesticides from field by runoff as well. Current crop production systems, which use plastic mulch (e.g., polyethylene), are less sustainable and may have harmful effects on the environment because of increased runoff volume and loading of pesticides in runoff from impervious surface.^[5]

TESTING METHODS TO ASSESS RUNOFF PROCESSES

Methods for the assessment of the runoff properties of pesticides are usually based on the modeling of the circumstances of a rainfall event following pesticide application. The laboratory modeling of processes on artificially prepared soil allows the precise measurement of the amount of rainfall and runoff water as well as pesticide concentration on soil and in water, and may contain a larger set of sampling sites. However, laboratory models may lack the real effect of evaporation, wind, and sunlight, and are, in most cases, carried out on artificially finite soil surfaces. A modeling of runoff could be performed under pilot plot and field conditions, which simulate real situations best. Such plots may contain crops, and reflect the cultivation

method that allows the evaluation of the effect of the cultures. Rainfall simulators allow the control of the amount of precipitate at smaller areas, and could be useful to study channel and sediment formation, and soil erosion as well. The measurement of the amount and chemical analysis of runoff water, sediment, and soil samples collected on field allows for the calculation of the mass balance of the pesticide, thus shedding insight into its adsorption, transport, and mobility properties. Runoff models, which are established at the laboratory, pilot plot, or field-scale representative for the soil properties of the geographical region, allow the calibration of expert systems established for the simulation of pesticide contamination and estimation of environmental quality. These systems are based on compound-specific physical-chemical data of the pesticide, together with functions derived from basic, compartment-level environmental phenomena influenced by local soil, geographical, and climatic properties. Time-dependent concentration profiles, together with the effect of the pesticide on ecosystems, allow risk assessment for the identification of locations and ecosystems.

RUNOFF MANAGEMENT PRACTICES

The application of planned pest management leads to reduction in erosion, pesticide losses, and contamination of the environment. This strategy includes: 1) avoidance of unnecessary pesticide applications; 2) use of targeted and economical applications; and 3) use of cultural or biological practices that substitute for or complement pesticide use. In addition, pesticide selection and crop management should be carried out based on site-specific needs. The first step of runoff management is a proper evaluation of soil structure, climatic and geographic conditions, type of the crop to be treated, and the pest type to be managed. In the second step, mathematical modeling could be performed to evaluate the ways of application, possible losses of pesticides by runoff and leaching, and its magnitude, spatial distribution, and pathways to the water bodies of the territory. Screening, research, and management computer models are effective tools to study the potential impacts of agrochemicals, and to make a comparison between the active ingredients to draft and to publicize a classification system based on the environmental sustainability of their use, and to defend the aquatic ecosystem at the field, watershed, and regional levels.^[5-8] Output from simulation models allows the long-term prediction of environmental quality; thus long-term policy-making regarding the use of xenobiotics may have an effect on natural ecosystems. To maintain environmental quality, appropriate limit concentrations as well as limit

emission loads for a given period of time might be established centrally or locally in certain regions, taking into account the special features of local environment. This also encourages the development of regionwide environmental management strategies of public and community interest. An important part of runoff management practices is the appropriate design of facilities serving the collection and draining of storm water and wastewater from areas treated or polluted with pesticides, and their regular maintenance. Vegetative buffer strips, artificial ponds, grass fields, and forested strip zones—aside from their role in the prevention of erosion, nutrient leakage, purification, and balancing storm water flows—act as reservoirs for collection of debris, filtration, and detention areas for decomposition of adsorbed and/or dissolved xenobiotics. A continuous monitoring of environmental quality at sites affected by pesticides is of paramount importance in runoff management. Monitoring allows the estimation of pesticide emission and loading to the environment, assessment of the efficiency of existing treatment systems, and detection of potential emission sources. Geographic information systems allow the identification of singularities, and provide useful feedback information for further actions to minimize the off-site impacts of agricultural activities.

CONCLUSION

In the concerted efforts to produce high-quality food and agricultural products, without compromising the state of human health and environmental quality, the collective handling of agricultural, economic, and environmental concerns is required. Proper application is based on local and regional factors influencing crops, production methods, pesticide use, and environmental impacts. Integrated pest management is a sustainable technology that involves the selection, integration, and implementation of pest control actions on the basis of predictable economic, ecological, and sociological consequences.

REFERENCES

1. Flury, M. Experimental evidence of transport of pesticides through field soil—A review. *J. Environ. Qual.* **1996**, *25*, 25–45.
2. Clark, G.M.; Goolsby, D.A.; Battaglin, W.A. Seasonal and annual flux of herbicides from the Mississippi River Basin to the Gulf of Mexico. *Environ. Sci. Technol.* **1999**, *33*, 981–986.
3. Konda, L.N.; Pásztor, Z.S. Environmental distribution of acetochlor, atrazine, chlorpyrifos and propisochlor

- under field conditions. *J. Agric. Food Chem.* **2001**, *49*, 3859–3863.
4. Piccolo, A. Interaction between organic pollutants and humic substances in the environment. In *Humic Substances in the Global Environment and Implications on Human Health*; Senesi, N., Miano, T.H., Eds.; Elsevier: Amsterdam, 1994; 961–980.
 5. Rice, P.J.; McConnel, L.L.; Heighton, L.P.; Sadeghi, A.M.; Isensee, A.R.; Teas Abdul-Baki, A.A.; Harman-Fetcho, J.A.; Hapeman, C.J.J. Runoff loss of pesticides and soil; A comparison between vegetative and plastic mulch in vegetable production systems. *Environ. Qual.* **2001**, *30* (5), 1808–1821.
 6. Verro, R.; Sala, S.; Auteri, D.; Finizio, A. Geographical Information System (GIS)-integrated application of distribution models and risk indexes for analysis of environmental compatibility of pesticides on a territorial basis: the case of the Lombardy region. *Ing. Ambient.* **2001**, *30* (10), 535–547.
 7. Ropke, B.; Bach, M.; Frede, H.G. Modeling pesticide input into surface waters in germany. In *Pesticide Behavior in Soils and Water*; BCPC Symposium Proceedings; British Crop Protection Council: Farnham, UK, 2001; 289–294.
 8. <http://www.lmnoeng.com/Hydrology/hydrology.htm> (accessed July 2002).

Safe Use of Pesticides: A Developing Country's Point of View

Catharina Wesseling

Central American Institute for Studies on Toxic Substances (IRET), Universidad Nacional, Heredia, Costa Rica

Clemens Ruepert

Fabio Chaverri

Central American Institute for Research on Toxic Substances (IRET), Universidad Nacional, Heredia, Costa Rica

INTRODUCTION

Pesticide use in agriculture has been promoted as an important tool for development for decades despite being an extremely hazardous technology for health and the environment. At the same time that a number of industrialized countries are undertaking significant steps to reduce pesticide use, developing countries are becoming a more important marketing target, and pesticide use is increasing in many developing regions.^[1]

For the last 20 years, international agencies, in collaboration with local governments, have used primarily 'safe use' approaches to reduce the risks of pesticide use in developing countries, the most important being the International Code of Conduct of the United Nations' Food and Agriculture Organization (FAO) and the "Safe Use Initiative" of the Global Crop Protection Federation (GCPF, formerly GIFAP, International Group of National Associations of Agrochemical Manufacturers; name recently changed to Croplife International). The main presumptions have been that pesticides are indispensable and, if properly handled, will not cause unreasonable harm.^[2,3] Collateral damage of pesticide use has been attributed primarily to insufficient regulation and the ignorance of pesticide users in developing countries, and it has been assumed that strengthening of regulations and education to users would lead to an acceptable level of pesticide safety.

LIMITATIONS OF "SAFE USE" STRATEGIES

Although the term "safe use" sounds safe, it really means the use of *dangerous substances* in the *absence of adverse health or environmental effects*. The "safe use" concept emphasizes the prevention of acute health effects among agricultural workers and farmers. The World Health Organization (WHO) International Program of Chemical Safety Classification of Pesticides

by Acute Hazard represents the most important regulatory guidelines for governments of developing countries that do not carry out independent risk assessments, and pesticides classified by WHO as non-hazardous are widely used in developing countries often without further questioning. For example, the possibly human carcinogens mancozeb, maneb, and chlorothalonil are frequently used in Central America.

The FAO Code of Conduct, first issued in 1985 and under revision since 1999, aims at the strengthening of pesticide registration and regulation by national governments. The Prior Informed Consent (PIC) procedure of the Code, an international agreement through the Rotterdam Convention since 1999, aims at controlling international commerce of particularly dangerous or obsolete pesticides. However, the voluntary nature of adherence to the Code and the small number of pesticides included in PIC, today only 24, limit the Code.^[2] Discussions about inclusion of WHO Ia and Ib (extremely and highly hazardous) pesticides or certain pesticides causing special problems in developing countries started about a decade ago but have not taken a concrete form. Industrialized countries continue to manufacture and formulate prohibited pesticides for export, mostly to developing countries.

The global stewardship program of the GCPF was initiated in support of the FAO Code of Conduct. Three pilot Safe Use Projects of GCPF conducted in Kenya, Thailand, and Guatemala are now being extended to other countries in Africa, Asia, and Latin America.^[3] It aims at enhancing collaboration between the private and governmental sectors in safe use training of pesticide users, distributors, agricultural extensionists, regulators, inspectors, and other sectors of the civil society including schoolchildren, teachers, and housewives. GCPF's statements that pesticide poisonings in Guatemala decreased as a result of the Safe Use Project have been challenged.^[4] The training materials assure the need, benefits, and safety of pesticides.

In Costa Rica, one of the world's largest agrochemical companies conducts a train-the-trainer program with officials of the Ministry of Health as multipliers. The use of industry's training materials on the community levels by these officials may result in promotion of pesticide use under the auspices of the Ministry of Health.

UNSAFE PESTICIDE USE AND HEALTH AND ENVIRONMENTAL CONSEQUENCES IN CENTRAL AMERICA

The key question is whether the safe use programs do in fact prevent pesticide-associated problems. Several comprehensive reviews on pesticide and health effects in developing countries exist,^[5,6] and there is no convincing evidence that "safe use" has substantially decreased adverse pesticide impact.

In Central America,^[5,6] pesticide use in the late 1990s increased as compared to the 1980s. Many heavily imported pesticides are of special toxicological concern including paraquat, mancozeb, terbufos, methamidophos, methyl bromide, carbofuran, aluminum phosphide, methyl parathion, copper arsenate, and aldicarb. Recent studies among farmers show increased knowledge and risk awareness without substantial changes in pesticide handling (Fig. 1). Almost 6000 poisonings were reported to the surveillance systems in the Region during 1998, but a realistic estimate exceeds 30,000. Cholinesterase depression and organophosphate residues in urine associated with such symptoms were documented among populations living close to aerially sprayed fields and family members of agricultural workers in Nicaragua and El Salvador. Chronic and delayed health effects from pesticide exposure documented in Central America include respiratory (paraquat), neurotoxic (organophosphates and DDT), dermal (paraquat and other pesticides), allergenic (chlorothalonil), genotoxic, and carcinogenic effects. In Costa Rica, each year several emergencies of massive poisonings events occur. Technical failure in a formulation factory resulted in a drift of

toxic gasses of terbufos to a school in 1996; a fire broke in a pesticide storehouse with mancozeb and organophosphates in the middle of the city of Alajuela in 2000; and a drift of a methamidophos application in a fern greenhouse poisoned children and teachers at an adjacent school in 2001.

Aerial spraying in crops like banana and rice still contaminates waterways and exposes populations (including children) living around these crops despite stricter regulations, the introduction of buffer zones, and the use of new technology. Several pesticides are frequently detected in surface water downstream from banana (fungicides), rice (herbicides) and pineapple (herbicides) plantations, and fern greenhouses (insecticides, herbicides). Surface water often drains into protected conservation areas like highly diverse wetlands, causing biological impact.^[7]

IS SAFE PESTICIDE USE AT ALL POSSIBLE IN DEVELOPING COUNTRIES?

If less dangerous alternatives are not available and safety conditions are adequate, the use of a hazardous substance may be temporarily justified, depending on expected benefits. A true safe use approach for pesticides in developing countries should consider first whether there is a real need for a certain pesticide, by examining local pest patterns and the accessibility of less dangerous alternatives (non-chemical or chemical). If so, it must be evaluated whether the use conditions guarantee that no health and environmental damage will occur. Regulatory authorities in developing countries need the capacity for local risk assessments as a basis for decision making, implying evaluation and integration of intrinsic toxicity data, exposure data, and considerations such as host susceptibility data, together with the socioeconomic, cultural, and legal entourage. Finally, once approved, the use and impact of hazardous substances must be closely followed up over time. However, the resources needed for such an approach exceed the economic and technical capacity of virtually any developing country.

Decision making in developing countries is subject to flaws. Human and technical resources are so limited that risk assessment is reduced to copying of international guidelines by FAO (Codex Alimentarius, PIC) and WHO (IPCS Classification by Acute Hazard), or of regulatory decisions by the European Commission or the U.S. Environmental Protection Agency (US-EPA). Decision making seldom looks at local or regional data on human health and environmental effects, or local circumstances of use, including hot humid climate, impossibility of purchase and use of protective equipment, general poverty, illiteracy, undernourished populations, very young and very old workers, lack of



Fig. 1 Farmers spraying pesticides on a potato field in Tierra Blanca de Cartago, Costa Rica, July 2000.

recycling facilities for pesticide containers, and an uncountable amount of further aspects unique to developing countries. Often, the interpretation of the toxicological, epidemiological, and exposure assessment data underlying risk management decisions in developed countries are clearly erroneous. Thus, in Central America, pesticides not registered for use but with a food tolerance in the United States, or pesticides that are regulated as restricted use pesticides (RUPs) by US-EPA, are routinely approved for a large variety of purposes and sold without restrictions.

Little or no effort is made to follow up the use and consequences of registered pesticides. Hazard or exposure monitoring, such as statistics on pesticide imports, exports, and use; or monitoring of residues in food, ground water, or environment is usually absent, deficient or not accessible to the public. No human, technical, or financial resources exist to carry out monitoring of adverse effects, including human, domestic animal, and wildlife poisoning surveillance systems; human health and ecotoxicological studies; and studies on economic consequences, such as costs of residue export retentions up to the complete bankruptcy of producers of certain crops due to pest resistance. Such studies are scarce, and even when local research institutions produce relevant data, these are seldom considered in local policy making.

The FAO Code of Conduct does not stimulate governments to develop their capacity for risk assessment with incorporation of local data in their decision making, but rather to follow the FAO guidelines. It takes years of deliberation and evaluation to include a new pesticide in the PIC list, and the influence of industry on decisions is disproportionate. GCPF has a clear conflict of interest with pesticide restriction or banning. There is no instance of industry forcing governments in developing countries to restrict their pesticides. Millions of dollars have been spent on drafting, discussing, negotiating, and evaluating the various international programs to promote safe use; however, the situation has hardly improved.

Preventive thinking about what a certain country can really handle in terms of risks is absent in pesticide regulation in developing countries. Usually, when problems emerge, authorities have no clue as to what to do and whom to charge for the damage. Despite hard experiences, still few thoughts are given to such issues in registration in Central America, and efforts are directed rather to harmonization of pesticide registration and regulations in line with the traditional safe use approach.

ALTERNATIVE APPROACHES TO PEST MANAGEMENT

Although “safe use of pesticides” has stirred some action in governments in developing countries and at

least put the problem on the agenda, it is evident, after 20 years of pushing it, that this approach will never reach its goals. Continuation of excessive focus on “safe use” seems irrational and will delay the development and implementation of other more effective strategies. Synthetic pesticide use in developing countries is not compatible with principles of sustainable development. In fact, “safe use” efforts encourage the use of pesticides and make countries desist from investing in sustainable agriculture.

Alternative approaches are needed, and successful examples exist. The FAO Code of Conduct, despite its prime safe use focus, has also promoted Integrated Pest Management (IPM) programs, which achieved important reductions in pesticide use especially in Southeast Asia, without endangering yield and economic return.^[2] Also, in Central America, IPM programs have been carried out by public and private agronomic research institutes on important crops like sugar cane, coffee, corn, citrus, and potato.^[8] IPM programs have proven to be more profitable than the conventional pesticide-based pest control after a transition period. The disadvantage of IPM is that it still recommends the use of some chemical pesticides. No use of pesticide is safe by definition, but more importantly, there is a real risk that the strategy turns into an attenuated safe pesticide use approach. GCPF, on one hand, commits itself to IPM strategy,^[9] on the other hand, it contradicts itself by advocating high-input and large-scale agriculture.^[10]

Organic agriculture does not use synthetic pesticides and integrates other principles of sustainable land use. Organic agriculture has been pointed out by industry as being a too radical strategy and to be unsustainable because of lower yields, which would force farmers to extend agricultural land use.^[10] However, studies are showing that organic agriculture can produce similar yields as traditional pesticide-dependent cropping and may be more profitable on the longer term due to sustainable methods.^[11,12] Failures in implementation, such as lack of extension and research services, are avoidable.^[8]

CONCLUSION

Safe pesticide use in developing countries does not seem feasible. The major ongoing programs for safe use of pesticides are costly and ineffective. After almost 20 years of pursuing “safe use,” import and exposure data as well as health and environmental research in Central America show how limited the impact of the safe use strategy has been. It is time to make profound changes in international and national agricultural policies and steer toward sustainable agriculture based on non-chemical pest management.

REFERENCES

1. Akhabuhaya, J.; Castillo, L.; Dinham, B.; Ekström, G.; Huan, N.H.; Hurst, P.; Pettersson, S.E.; Wesseling, C. *Multistakeholder Collaboration for Reduced Exposure to Pesticides in Developing Countries. Recommendations to Sida with Particular Reference to Costa Rica, Tanzania and Vietnam*; Stockholm, 2000 http://www.pan-uk.org/briefing/sida_files/index.htm.
2. <http://www.fao.org/ag/AGP/AGPP/Pesticid/Default.htm> (accessed August 2001).
3. <http://www.gcpf.org> (accessed August 2001).
4. Murray, D.L.; Taylor, P.L. Claim no easy victories: Evaluating the pesticide industry's global safe use campaign. *World Dev.* **2000**, *28* (10), 1735–1749.
5. London, L.; de Grosbois, S.; Wesseling, C.; Rother, H.A.; Kisting, S.; Mergler, D. Pesticide usage and health consequences for women in developing countries: out of sight, out of mind? *Int. J. Occup. Environ. Health* **2002**, *8* (1), 46–59.
6. Wesseling, C.; Aragón, A.; Castillo, L.; Corriols, M.; Chaverri, F.; de la Cruz, E.; Keifer, M.; Monge, P.; Partanen, T.; Ruepert, C.; van Wendel de Joode, B. Hazardous pesticides in Central America. *Int. J. Occup. Environ. Health* **2001**, *7* (4), 287–294.
7. Castillo, L.E. *Pesticide Impact of Intensive Banana Production on Aquatic Ecosystems in Costa Rica*; Stockholm University: Stockholm, Sweden, 2000. Doctoral dissertation.
8. García, J.E. Propuestas de Solución a la Problemática de los Plaguicidas. In *Introducción a los Plaguicidas*; García, J.E., Ed.; Editorial Universidad Estatal a Distancia: San José, 1997; 287–352.
9. http://www.gcpf.org/library/position_papers/new/45.html (accessed August 2001).
10. http://www.gcpf.org/library/position_papers/new/42.html (accessed August 2001).
11. Reganold, J.P.; Glover, J.D.; Andrews, P.K.; Hinman, H.R. Sustainability of three apple production systems. *Nature* **2001**, *410*, 926–930.
12. Boyce, J.K.; Fernández, G.A.; Fürst, E.; Segura, B. *Café y el Desarrollo Sostenible: Del Cultivo Agroquímico a la Producción Orgánica en Costa Rica*; Editorial Fundación UNA: Heredia, Costa Rica, 1994.

Safe Use: Farmers' Association Point of View

Alarik Sandrup

Ministry of Agriculture, Stockholm, Sweden

INTRODUCTION

Despite the low degree of pesticide used in Sweden, it still causes a number of problems. These problems can take the form of inadvertent spreading to other crops, ecosystem imbalance, and residue in surface and ground-water. Earlier, there had also been health problems among farmers, but this has been reduced as the development of pesticides, protective equipment, and techniques has evolved in combination with the increased awareness of farm workers.

More stringent legislation was applied in Sweden in 1997 in an attempt to reduce environmental problems. This brought with it a debate on how detailed the legislation should be, and it was finally resolved that excessively detailed legislation would not be efficient. Instead, the Federation of Swedish Farmers (Lantbrukarnas Riksförbund, LRF) took the initiative of introducing a campaign called Safe Pesticide Use to promote information and education directed at farmers, consultants, sales people, and others who worked with pesticides. This initiative was taken together with the National Chemical Inspectorate, the Swedish Board of Agriculture, the Swedish Farmers' Supply and Crop Marketing Association, and the Pesticide Producers Organisation.

LRF AND THE SWEDISH APPROACH

Since the beginning of the 1990s, LRF has been working actively with issues and problems concerning sustainability, ethics, and food safety. LRF's fundamental standpoint in this commitment is that problems can be solved in the long term only by involving everyone who is affected and not just by force and central control. Legislation may be necessary to establish a framework, but to "fine tune" the agri-environmental efforts, it must be possible to consider other influencing factors, individual conditions, new concepts, etc. Cost-efficiency must be the guiding light. This can only be achieved if the implicated players are committed and feel that they are participating in the problem. This bottom-up perspective has also played a central role in the general commitment to Agenda 21, which is considered to have been successful in Sweden.

Important components of LRF's environmental strategy include: a) cooperation with authorities and other players; b) consumer perspectives instead of producer perspectives; c) staying one step ahead and approaching problems in good time; and d) striving for openness and dialogue with all affected parties, both inside and outside agricultural circles.

SWEDISH AGRICULTURE—EXTENSIVE AND ENVIRONMENT-FRIENDLY

Swedish agricultural production varies greatly due to the extent of the country and its varying climatologic and geographic conditions. The area of arable land totals approximately 2,750,000 ha, which only accounts for around 7% of the total land area. Family-owned and -managed agricultural farms combined with dairy production as the main source of income are the most common type of farms in Sweden. The average area of a farm in Sweden is 34 ha.

Plains farming is carried out in some of the southern and the central regions and is mainly concentrated on growing crops and, in some cases, also pig farming. Ley farming is practiced on 36% of arable land, and its produce is used as feed in milk production, cereal is grown on 42%, and 12% is set aside. The remaining 10% of arable land is used for potatoes, sugar beet, leguminous plants, and oleiferous plants. About 12% of the total area is used for organic cultivation, which among other things means that neither chemical pesticides nor industrial fertilizers may be used.

The crops harvested vary; an average harvest of winter wheat is 6000 kg/ha, and, for spring grain, it is around 4000 kg. Spread over the whole area, an average of 80 kg nitrogen fertilizer and about 0.6 kg active substance pesticide per hectare arable land is used. Farming uses almost 1700 t of active substance pesticide annually, of which about 75% is herbicides (including glyphosate), 15% is fungicides, and 4% is insecticides.^[1] The remaining 6% comprises seed dressing preparations and growth regulation.

The sale of pesticides dropped from around 4500 t in early 1980s to just over 1500 t in the mid-1990s. After Sweden's entry into the European Union (EU) in 1995 and its adaptation of the Common Agricultural

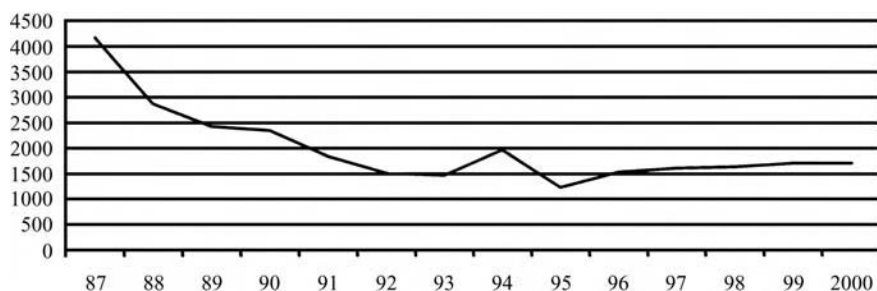


Fig. 1 Tons of active substance pesticide sold to Swedish farms in 1987–2000. The drop is partly due to the introduction of low-dose pesticides. Variation in sales in 1994–1995 is due to increase in environment levies on pesticides introduced in 1995.

Policy (CAP), sales rose to today's level of almost 1700 t (Fig. 1).

Naturally, the EU's agricultural policies affect Swedish farming in several ways. The increase in sales of pesticides after entering the EU in 1995 is deemed to be largely caused by an increased intensity. Grants to Swedish farming in 2000 amounted to SEK 8.5 billion (\$780 million), of which the largest portion came from the EU. A little over 30% of this sum was used on various environmental measures, e.g., organic production and measures to reduce nitrogen emissions.

SAFE PESTICIDE USE CAMPAIGN

The campaign has distributed information in the form of brochures, courses, information sheets, advertisements in agricultural magazines and on a web site (www.lrf.se/sv). Brochures have been distributed via retailers, consultants, and at compulsory certification courses, and they can be ordered from the web site. The courses have been arranged locally in a large number of venues and have been attended by farmers, consultants, salesmen, and authority representatives. The information sheets have been sent to all 30,000 farmers in the country who are qualified to use pesticides. All the information has been available free of charge and was financed by the Swedish government and the EU.

The information campaign has focused mainly on the following areas:

Filling and Cleaning

These processes are the point sources of emissions that can spread via drain water or to the groundwater. The campaign recommends these processes to be carried out, firstly, on a biological bed or on a slab with collecting possibilities and, secondly, on biologically active ground.

Safety Distance to Ground Transport

In order to reduce surface run-off in conjunction with filling and cleaning, a safety distance to avoid ground

transport of 30 m is recommended to wells, ditches, lakes, and waterways.

The following minimum safety distances are recommended in order to reduce the risk of surface runoff when spraying: 1 m to wells and ditches, 6 m to lakes and waterways, and 12 m to drinking water wells.

Safety Distance for Wind Drift

In order to protect water, the surrounding ecosystem, and other crops, the safety distance must be adjusted to accommodate for wind speed, temperature, dose, and the sprayer that is used. Special tables have been produced to this end with recommended safety distances from 2 to over 50 m.

Other Information

The campaign also included information concerning preventive measures and the bases for decision making, the storage of pesticides, weed control in farmyards, handling of empty packaging, and personal protective equipment.

CAMPAIGN RESULTS

The results of an information campaign can be evaluated or measured in several ways. Perhaps the most interesting finding is whether pesticide concentrations in water are reduced in conjunction with the educational and informative materials disseminated. However, obtaining these findings requires a good deal of resources, and the causality is not always distinct. Neither is there much data concerning pesticide concentration in Swedish waters, which makes it hard to obtain reference values. Water samples that have been taken so far do not form part of any systematic sampling or study, which is why it is difficult to make any conclusions from the sporadic and few findings that have been made. However, Jenny Kreuger at the Swedish University of Agricultural Sciences has made a study that can be linked to the result of recommended routines.^[2] The study, which was carried out

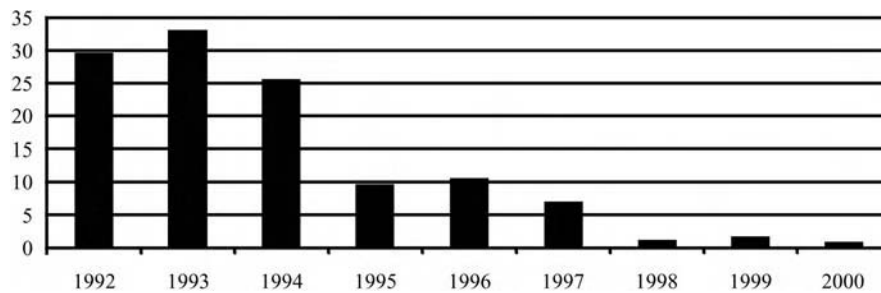


Fig. 2 Average concentration of total pesticides in May–September (May–June only in 1993). Average concentration weighted for time in surface water ($\mu\text{g}/\text{l}$). Voluntary consultations and information in accordance with Safe Pesticide Use were initiated in the mid-1990s. (From Ref.^[2])

in a dense agricultural area in southernmost Sweden (the Vemmenhög project), found that the concentration of pesticides is reduced considerably when the routines recommended in the campaign were applied (Fig. 2).

An alternative method of measuring the results of an information campaign is to check how well the message has reached the target group. Safe Pesticide Use has achieved this by engaging an independent institute to carry out a poll among a representative group of 1000 farmers. The poll showed that younger farmers running large farms in agriculture-intensive regions have received the campaign message best. Considering that this group is often well educated, that they run professional and intensive crop farming and have often received environmental information before, this result is perhaps not so surprising. Awareness of the informational material has increased over the 3 years the study was carried out. Among the farmers interviewed who used chemical pesticides in 2000, only 19% had not read the information or were aware of the campaign. With respect to changes in routines, depending on the routine in question, 24% to 56% had carried out improvements due to the campaign. Thirty-six percent of the interviewees stated that they had not changed their routines as they already considered them to be sufficient.

Despite the campaign having largely reached out with its message, there is still a good deal of change that must take place within Swedish agriculture. For example, there are only 25% of farmers who have at least a 30-m safety distance to wells, ditches, lakes, and waterways when refilling. There is also some work left to do with regard to selecting a site where the filling of the sprayer should take place. Forty-five percent fill their sprayers in locations recommended in the campaign, i.e., on a biological bed or on a slab with collecting possibilities. As many as 20% are still filling their sprayers on farmyards, which is considered to be a

definite hazard to the environment. However, great improvements have been detected during the campaign.

CONCLUSION

In order to attain the goal of a sustainable food production, the means used to achieve this goal must also be sustainable. This means that the players who must change their behavior must be given the required knowledge in order to understand the entire problem and to know how to solve it. The key to this end is the individual's knowledge, the individual farmer in this case. Knowledge in combination with the opportunity to take responsibility will hopefully lead to involvement and a will to solve the problems mankind occasionally causes. As environmental problems become more complex, the likelihood of solving them through central means of control also becomes smaller.

Over the years, the Safe Pesticide Use campaign has been active; it has shown that conveying knowledge in a voluntary manner has had a far-reaching positive effect. This does not mean that legislation and other regulatory means should be abolished, but that the biased belief in legislation and central means of control that often characterizes environmental efforts may need to be reconsidered.

REFERENCES

1. *Bekämpningsmedel i Jordbruket 1999, MI 31 SM 0001*: Statistics Sweden (SCB): Stockholm, Sweden, 1999.
2. Kreuger, J.; Nilsson, E. Catchment scale risk-mitigation experiences—key issues for reducing pesticide transport to surface waters. In *Pesticide Behaviour in Soils and Water*; Walker, A., Ed.; BCPC Symposium No. 78; 2001; 319–324.

Safe Use: Industry Point of View

John L. Aston

Safe Use Project, CropLife International, Nairobi, Kenya

INTRODUCTION

The use of crop protection chemicals expanded rapidly into the developing countries of the world some 50 years ago following the discovery of organochlorine and organophosphate groups of insecticides. This rapid proliferation brought about problems of environmental pollution and unsafe handling of these products by untrained, poorly educated workers.

The crop protection industry and national governments have reacted to these problems by regulating use, phasing out more toxic and persistent molecules, and spending heavily on product stewardship and safe use training. Safe use training has been taken up by many donor agencies, Government extension services and non-governmental organizations (NGOs) around the world, and combined efforts have already led to major advances in the safe handling of these products.

The industry will certainly continue to invest heavily on safe use training as it recognizes the need for ongoing education and is fully aware of the dangers of complacency.

SAFE USE

The use of agricultural chemicals started centuries ago; arsenicals were reported in use as rodenticides in the 16th century.^[1] Other early introductions included the use of mercury compounds as seed dressings, and copper sulphate and calcium hydroxide (Bordeaux mixture) as preventive fungicides.

With the exception of Bordeaux mixture, these compounds have long been superseded by less toxic and more effective products. A major leap in the development of crop protection chemicals came in the 1940s when DDT and other organochlorines were introduced, shortly followed by organophosphate insecticides.

Since the 1940s, many new chemical groups have been introduced and have made enormous contributions in terms of safeguarding of food production and control of diseases such as malaria. Although millions of lives have been saved, the World Health Organization recently reported that more than 3 billion people are now malnourished.

Crop protection chemicals have also played a major part in ensuring food security in the developed world

where there is an abundance of good-quality, safe, and reasonably priced food. However, the rapid development of the industry since the 1940s has brought about its own problems.

The extensive, and sometimes excessive, use of organochlorines became environmentally unacceptable despite saving millions of lives, and the group has now been largely replaced by less persistent compounds. Only the much maligned DDT is still used in any quantity, with its use restricted to the public health sector in some 20 countries.

Public concerns on safety became more apparent with the advent of organophosphate and carbamate insecticides. Rachel Carson's famous scientific treatise, "The Silent Spring," echoed these concerns very concisely. Nevertheless, the major benefits, in terms of plant protection, better yields, and increased food security, were all too clear for the world to see. Unfortunately, rapid increases in product use, particularly in developing countries, outstripped industry efforts to adequately educate those handling and applying its products. Occupational poisonings were recorded alongside accidents and suicides. At the same time, the introduction of new groups of herbicides and fungicides had far less serious consequences because of their more favorable toxicological profiles.

Public concern relating to the use (and misuse) of these chemicals led quickly to the regulation of the industry and to the introduction of product registrations.

Basic safety requirements were first introduced by legislation in the United States. European countries subsequently followed suit with a series of compulsory tests to ensure that new products reached the market only after a series of laboratory and field trials. Over the years, these tests have increased in number and detail, so that any new crop protection chemical reaching the market today will have passed over 120 stringent trials, covering most known safety risk factors. On top of this, the chemical must also pass field bio-efficacy trials. Today, crop protection products are the most regulated group of chemicals in the world. But registration is only part of the story; the industry must also provide highly detailed labels for their products, which are designed to cover all use aspects, including safety measures and instructions for safe disposal of products and containers.

As far back as 1985, the industry and the United Nations Food and Agriculture Organization (FAO) have worked together to produce an International Code of Conduct for all aspects of agricultural chemical usage, from the production of the molecule to the safe use by farmers.^[2]

However, long before this, the major producing companies had recognized the need to ensure that products were used safely everywhere, and safe use practices were being heavily promoted by individual companies.

Occupational health and safety standards were implemented at all production points to protect workers against exposure to dust, gases, and liquids involved in the manufacture of active ingredients and formulated products. Standards and guidelines for safe storage, transport, and disposal have also been developed and compliance has been encouraged worldwide.

Ethical marketing and advertising standards have been defined and are increasingly incorporated into government legislation in many countries.

Strict product labeling legislation is enforced in most countries today, and the industry position, that all agricultural chemicals are safe to use if the label instructions are followed, has stood the test of time.

Unfortunately, because of lack of education and complacency, label recommendations are not always followed as they should be. To address the issues of literacy and the diversity of local languages, the industry developed the pictogram concept^[3] designed to inform rural farmers of the need to take precautions in simple pictorial form. The principle has been extensively adopted by governments worldwide.

Color coding of labels, developed by the FAO,^[4] has also been adopted and extensively legislated. A color band on labels (red for toxic products, yellow for moderately toxic products, blue for slightly toxic products, and green for relatively harmless products) is now widely accepted worldwide as a standard warning measure to denote the degree of care users must take when handling any specific product.

The research and development-based crop protection companies have cooperated and worked with governments to phase out more toxic compounds. Many organophosphate chemicals that were key components of the early crop protection decades have either been withdrawn, or are being replaced by products with much more favorable toxicological profiles and lower dosages.

Older herbicides such as triazines, which are still major market components, are slowly making way for more modern compounds that cause minimal threat to water sources. The industry's research and development efforts continue to discover and develop better alternatives, albeit at high cost, for end users.

In the last two decades, the main research and development-based companies have greatly increased effort and expenditure on product stewardship.^[5] All companies now have large and active departments working solely on safety issues.

Apart from cooperating with legal and registration authorities worldwide, companies have initiated large-scale training and education programs, both general and product-specific. Particular efforts have been made to ensure that end users in developing countries are made fully aware of potential risks when handling chemicals.

Over the years, the Brussels-based CropLife International (representing the plant science industry) has produced educational brochures on all important aspects of safe product use. These brochures are distributed free of charge worldwide in large numbers.

Safe Use Pilot Projects were set up 10 years ago in Kenya, Thailand, and Guatemala to determine the best training methodology to reach large numbers of small-holder farmers. Since then, the projects have trained over 1.5 million farmers in the three countries.

Safe use initiatives are now in place in some 40 countries worldwide and training messages are being taken up increasingly via donor funding, nongovernmental organizations, and Ministry of Agriculture extension service teams; examples of countries include Sri Lanka, Bangladesh, Vietnam, Nicaragua, Argentina, Brazil, Sudan, Lebanon, and Tanzania.

Training and education have also been extended widely to remote rural communities via radio programs and, to a lesser extent, via television and more modern electronic media presentations. It must be noted that in many developing countries, radio is still, by far, the most popular medium. Farming programs are always avidly listened to as technical education is hard to access in these situations.

Intensive training and educational work have been demonstrated to bring major and sustainable improvements in the safe use of crop protection chemicals, and the private sector and other stakeholders need to work together to continue achieving significant sustainable improvements in standards. Young people continuously enter agricultural work so there is a constant and increasing demand for education and training programs.

As an example, consumer concerns over food safety have led to the need to ensure that food exports from developing countries are produced using good agricultural practice and comply with appropriate worker welfare schemes. Rigid adherence to safety intervals between the last pesticide application and harvest has to be demonstrated to ensure that food reaches the consumer well within internationally accepted residue limits. New European Union legislation to this effect is now being implemented.

Poisoning incidents related to the use of crop protection chemicals have fallen in recent years. Dr. E. D. Richter of the *Encyclopedia of Pest Management* reports that there are 26 million nonfatal human pesticide poisonings each year. Already at a low level in the developed world, cases of occupational poisonings in most developing countries are declining, as older, more toxic products are replaced by less toxic groups that are active at lower rates. Government legislation in many countries has greatly assisted this cause as more highly toxic products have been banned or had their use greatly restricted.

By far, the most common cause of pesticide poisonings in developing countries relates to deliberate actions. Statistics from several countries indicate that up to 80% of poisoning incidents are deliberate, mostly suicide attempts, most of which are made in rural areas. The crop protection industry has assisted medical services in many developing countries to set up poison treatment centers and has distributed antidotes and literature widely.

On a quite different theme, the correct use of crop protection products has led to significant yield increases in all crop areas. Uninformed criticism of environmental damage caused by these chemicals is common. What most environmentalists do not understand is that, without these products, vast areas of natural habitats, forests, wetlands, and the like would have to be put to the plough to feed our world's ever-growing population. In a world without crop protection chemicals, millions of hectares of wildlife habitats would be lost. Without crop protection products, the destruction of habitats of endangered species would take place at an even more rapid rate.

Chemical control will remain an integral component of agricultural production for the foreseeable future,

but must be used safely, judiciously, and responsibly in combination with other means of protection. Our food production industry is committed to fully complying with principles of integrated pest management,^[6] which is, itself, a fundamental pillar of agriculture.

REFERENCES

1. Worthing, C.R., Ed.; *The Pesticide Manual*, 8th, Ed.; British Crop Protection Council: Thornton Heath, UK; 33.
2. Food and Organisation of the United Nations; Rome; 1985.
3. *Pictograms for Agrochemical Labels*; GIFAP 1988: Avenue Louise 142, Brussels B-1050, 19881270/20M/3.88/ENG/KRN.
4. *Good Labelling Practice for Pesticides*; FAO: Rome, March, 1985.
5. [http://www.croplife.org/library/position papers](http://www.croplife.org/library/position_papers)
6. <http://www.ecpa.be>

BIBLIOGRAPHY

- Guidelines for Emergency Measures in Cases of Crop Protection Product Poisoning*; CropLife International (formerly GCPF): Brussels, Belgium. <http://www.gcpf.org>.
- Guidelines for Personal Protection When Using Pesticides in Hot Climates*.
- Guidelines for the Safe and Effective Use of Crop Protection Products*; CropLife International (formerly GCPF): Brussels, Belgium. <http://www.gccpf.org>.
- Oerke, E.-C.; Dehne, H.-W.; Schonbeck, F.; Weber, A. Crop production and protection. In *Estimated Losses in Major Food and Cash Crops*; Elsevier Science B.V.: Amsterdam.
- Safe Use Pilot Projects*; GCPF: Brussels, Belgium, 1998. <http://www.croplife.org>, e-mail: gcpf@pophost.eunet.be.

Safe Use: Regulator's Point of View

Nguyen Huu Huan

Department of Plant Protection, Ministry of Agriculture and Rural Development,
Ho Chi Minh City, Vietnam

INTRODUCTION

Pesticides are biocides by design and have high potentials of affecting non-target organisms. For these products to be efficiently used, with minimal effects on human health and environment, countries need to adopt policies that will protect consumers and introduce programs that will encourage wise use. A regulator's important task is to ensure that products in the market have sufficient safety features. Pesticides with high risks to human health and the environment should not be allowed. It is also the responsibility of importers and manufacturers to ensure safety and adhere to country policies. In addition, a regulator should introduce programs that will improve consumer awareness on pesticide risks and prevent misuse.

SAFE USE: A REGULATOR'S POINT OF VIEW

Since 1940, pesticide use worldwide has been increasing from a balanced use to excessive use, and, finally, pesticide crisis. The total annual global pesticide use is about 2.5 million tons, which is mostly concentrated in the four biggest markets: America (20% of the world market), Japan, France, and Brazil. Developing countries with large and fast-growing markets for pesticides are Argentina, China, India, Indonesia, Mexico, Pakistan, the Philippines, and Vietnam. The total value of the world pesticide market is about \$30 billion per year, of which the proportion of the developing countries increased from 22% in 1978 to 31% in 1997 (G. Ekstrom, personal communication, 2000).

Pesticides have contributed to ensuring agricultural productivity, but at the same time have been creating risks to human health and the environment. In response to protests by public interest groups who have called attention to those risks, the Global Crop Protection Federation launched a Global Safe Use Campaign in 1991 to train pesticide users in developing countries. However, the analysis of the effectiveness and impacts of the Safe Use campaign for reducing pesticide risks and improving users' awareness has shown many contradictions and raised many issues.^[1] Dr. Russ Dilts, Food and Agriculture Organization (FAO) Regional Community Integrated Pest Management

(IPM) Coordinator, said that although most pesticide producers say that they make many efforts for the safe and effective pesticide use, it is estimated that there are about 25 million cases of pesticide poisoning every year, and most of the victims are from developing countries; besides that, about 220,000 people in the world die every year from pesticide poisoning.

Farmers' pesticide use depends on many factors, such as climatic conditions, cropping pattern, crop cultivars, pest pressure, farmers' income, ratio of pesticide costs/price of agricultural products, management methods, government regulations, farmers' education, traditional practices, farmers' attitude and perceptions, etc. Pesticide producers and governmental pesticide managers believe that pesticides are safely used and sprayed at appropriate crop development stages, at appropriate times. The reality is absolutely different in the fields and in hamlets and villages. Dr. Russ Dilts and a BBC video group, following the "Toxic Trail" from Thailand, highlighted one typical example in Cambodia. Pesticides classified as Ia "extremely hazardous" by the World Health Organization (WHO), e.g., methyl marathion, monocrotophos and mevinphos, which have been banned or restricted in many developing countries, are sold all over Cambodia. Poor Cambodian farmers are using these toxic pesticides regardless of warning. "Normal use" in the field is very different from the "safe use" recommendations of pesticide companies. Thus farmers are actually showered with different toxic cocktails.^[2]

Therefore the phrase "safe use" should be replaced with "pesticide risk reduction" from the regulator's point of view. It means to reduce pesticide risks to human health, non-target species, and the environment by reducing the dependency on chemical pest control methods, and also by reducing the amount and toxicity of pesticides used when pest control intervention with some sort of pesticide is necessary.

Reduce Pesticide Risk to Human Health, Non-Target Species, and the Environment

Organization for Economic Cooperation and Development (OECD) member countries have developed pesticide indicator tools to help government agencies to wisely manage pesticides, evaluate pesticide policies,

Table 1 Pesticide active ingredients banned and restricted in Vietnam, 1992–2000

| Year | Number restricted | Number banned |
|------|-------------------|--|
| 1992 | 14 | 20 |
| 1994 | 15 | 22 banned; additional five pesticides banned on rice: carbofuran, monocrotophos, methamidophos, endosulfan, and phosphamidon |
| 1996 | 21 | 22 banned; three banned for import: methamidophos, monocrotophos, and carbofuran |
| 1998 | 19 | 23 |
| 2000 | 27 | 26 |

Source: Plant Protection Department, MARD.

and measure pesticide risks to human health and the environment. All OECD member countries have regulations and a registration system to evaluate pesticides before allowing their sale to avoid unacceptable risks to the environment and human health.

The Malaysian government's 1974 Pesticides Law and a series of corresponding pesticide management regulations provide a good model for developing countries that wish to reduce pesticide risks.^[3] After coming into force, this regulatory system has been constantly monitored and strengthened.

Vietnam's State Law of Plant Protection and Quarantine was issued in 1993. So far, government pesticide regulations have significantly reduced the application, both in terms of products and the total amount imported, of a number of restricted-use pesticides that carry high risks to human health and the environment (Tables 1 and 2).

In addition, there are other policies and measures, such as pesticide taxes, that can be used to minimize the effects of pesticide use on human health and the environment.^[4]

Reduce Dependency on Chemical Control Methods

The 1999 OECD/FAO Workshop on IPM and Pesticide Risk Reduction emphasized the important role of IPM in reducing pesticide risks:

- Reduces dependency on pesticides and encourages alternatives.
- Encourages using less-risky pesticides, when pesticides are necessary.
- Prevents pest development, based on good crop management and protection by the natural enemies of pests and plant diseases.

- Improves farmers' awareness about pests, natural enemies, and the field ecosystem.

Thus IPM is considered to be a good approach for pesticide risk reduction. Back to the pesticide-use situation in Cambodia, the IPM program implemented by FAO helps farmers to better understand pesticides and their acute effects on human health, such as vomiting, trouble in walking, dizziness, burning eyes and skin, muscle cramps, and shortness of breath. Some farmers experience these acute symptoms after a spray operation. After IPM training, these farmers reduce their pesticide use and experience less-acute symptoms of pesticide poisoning.^[2]

In Indonesia, the National IPM Program helped IPM farmers to reduce their pesticide use. They established an organization of farmers that sell organic rice (produced without chemical pesticide and chemical fertilizer) in many shops in their community.

In Vietnam, rice farmers have reduced pesticide use from 3 to 4 times per season to once per season during the period 1992–1997.^[5] At the same time, IPM helps farmers to reduce costs of medical care for acute pesticide poisoning,^[6] etc.

Reduce Pesticide Use

Experiences from three northern European countries—Sweden, Denmark, and Netherlands—in the last two decades of the 20th century are valuable lessons for developing countries. The governments of these three countries developed strategic and sustainable policies to reduce total domestic pesticide use by 50%.^[7] Sweden's "Pesticide Risk Reduction Program" started in 1986 with the aim of reducing pesticide use by 50% in the first 5 years (1986–1990). After achieving that goal, the Swedish government continued the policy in their following plan. After 15 years of implementing this sustainability policy, Sweden has greatly reduced

Table 2 Restricted Pesticides Imported into Vietnam, 1991–1998 (Tons)

| Year | Total imports | Restricted pesticides | Percentage (%) |
|------|---------------|-----------------------|----------------|
| 1991 | 20,300 | 7,500–8,000 | 36.9–39.4 |
| 1992 | 23,100 | 7,500–8,000 | 32.5–34.6 |
| 1993 | 24,800 | 7,500–8,000 | 30.2–32.3 |
| 1994 | 20,380 | 3,000 | 14.7 |
| 1995 | 25,666 | 3,000 | 11.7 |
| 1996 | 32,752 | 3,000 | 9.2 |
| 1997 | 30,406 | 2,500 | 8.2 |
| 1998 | 30,000 | 1,500 | 5.0 |

Source: Plant Protection Department, MARD.

Table 3 Reduction in pesticide quantities and acute pesticide-related health risks, Sweden, 1981–2000

| Period | Sold quantity of active substance (tons) | Reduction percentage of baseline period | Acute toxicity equivalents | Risk reduction percentage of baseline period |
|----------------------|--|---|----------------------------|--|
| 1981–1990 (baseline) | 23,000 | — | 38,000 | — |
| 1991–1995 | 9,000 | 61 | 11,000 | 71 |
| 1996–2000 | 9,300 | 60 | 8,700 | 77 |

Source: Ref.^[8].

both the amount of pesticide used and the risks to human health and the environment (Table 3).^[8] Denmark's "Action Plan to Reduce Pesticide Application" aimed to reduce by 50% both the number of spray events and the total amount of applied pesticides. During the first 5 years of the plan (1986–1990), they reduced the amount of active ingredient used by 25%; by 1997, the number of spray events and the total amount used were both reduced by 50%. The Netherlands "Multiyear Crop Protection Plan" also met its 1995 target of a 35% overall reduction in the use of pesticide active ingredient and by 50% by the year 2000.^[7]

The FAO's regional IPM programs in different crops greatly contribute to minimize pesticide application in the field. The FAO Community IPM Program in Asia trained a hundred thousand extension workers and a million farmers in Farmer Field Schools from 1998 to 2002, improving farmers' skills at agro-ecosystem management. Knowledge about the crop field ecosystem can be applied to the analysis and management of the wider community and family ecosystems.^[9]

CONCLUSION

The phrase "safe use" is the voice of pesticide manufacturers. It envisions a relationship between consumers and pesticide manufacturers. Manufacturers should be responsible for providing users with guidance on how to safely use their products, as in the speech of Prime Minister Phan Van Khai of Vietnam on November 13, 2000, with the Ministry of Agriculture and Rural Development (MARD): "MARD and Provincial People's Committees have specific regulations, ... to assign responsibility to organizations and individuals that produce and sell seeds, and agricultural inputs, to ensure that these products do not harm producers."

From the pesticide managers' angle, safe use means reducing pesticide risks to human health and the environment, with emphases on the following:

- Improve awareness of community and pesticide users that "Pesticides are poisons."
- Strengthen training activities to train managers, leaders, extension workers, and farmers on IPM and sustainable agro-ecology.
- Finalize strategic policies to reduce the amount of pesticide used, replacing highly toxic pesticides with less-toxic pesticides.

REFERENCES

1. Murray, D.L.; Taylor, P.L. Claim no easy victories: Evaluating the pesticide industry's global safe use campaign. *World Dev.* **2000**, *28* (10), 1735–1749.
2. Toxic Trail@attglobal.net and Television Trust for the Environment (TVE) TVE-dist@TVE.org.uk.
3. Tan, S.H. Towards Reducing Risks of Pesticides—Some Malaysian Experiences, Rice IPM Conference—Integrating Science and People in Rice Pest Management, Kuala Lumpur, Malaysia, November, 18–21, 1996.
4. *Report of The OECD/FAO Workshop on IPM and Pesticide Risk Reduction*; OECD Series on Pesticide Number 8, ENV/JM/Mono (99) 7, Paris, 4/1999.
5. Huan, N.H.; Mai, V.; Heong, K.L.; Escalada, M.M. Changes in rice farmers' pest management in the Mekong Delta, Vietnam. *Crop Prot.* **1999**, *18*, 557–563.
6. Dung, N.H.; Dung, T.T.T. *Economic and Health Consequences of Pesticide Use in Paddy Production in the Mekong Delta, Vietnam*; EEPSEA: Singapore, 1999.
7. Matteson, P.C. The "50% pesticide cuts" in Europe; A glimpse of our future? *Am. Entomol. Winter* **1995**.
8. Ekstrom, G.; Bergkvist, P. Persistence pays—Lower risks from pesticide in Sweden. *Pestic. News* December **2001**, *54*, 10–11. PAN-UK.
9. PAC Report. The FAO Programme for Community IPM in Asia, Programme Advisory Committee (PAC) Meeting, Ayutthaya, Thailand, November, 26–28, 2001.

Satellite Imagery in Pest Management

John LeBoeuf

AgriDataSensing, Inc., Fresno, California, U.S.A.

INTRODUCTION

New visual information technologies, derived from satellite sensors and displayed in digital formats, are being used for pest management in production agriculture. There are practical applications associated with the use of technology developed by the aerospace industry. Remote sensing of vegetation is aimed at reflectance of energy in the near infrared portion of the electromagnetic spectrum and absorption of energy in the visible range of light. Imagery collected during the growing season offers progress reports on plant health. When turnaround time of imagery is less than 72 hr, from the time of data acquisition until delivery into the hands of end-users, satellite data can be used for pest monitoring.

With various satellite and aerial platforms in operation from public and private sectors, a review of image capabilities will be examined to illustrate data content. Change detection maps, green vegetation maps, and color infrared images are used to detect plant stress, which is a major strength of remote sensing technology. This document will identify some pest management uses of remote sensing imagery, while it will also explain what it should not be expected to do.

SATELLITE IMAGERY IN PEST MANAGEMENT

Sensor Generated Information

In the mid-1980s, equipment with tractor mounted on-the-go sensors generated field-specific information that identified variability in much greater detail than farmers had previously known about. Because on-the-go sensors are in direct contact with soil, these types of sensors are not considered in the same context as remote sensing, which traditionally meant that information was gained from sensors in remote locations that were not in direct contact with an object. New visual information technologies, derived from satellite sensors and displayed in digital formats, are being used for pest management in production agriculture. There are practical applications associated with the use of technology developed by the aerospace industry. Remote sensing imagery collected during the growing season offers progress reports on plant

health. When turnaround time of imagery is less than 72 hr, from data acquisition until delivery into the hands of end-users, satellite data can be used for monitoring with some excellent applications for pest management.

Precision Agriculture

As a suite of technologies became available to innovators, a new management strategy that combined information with decision-making became known as precision agriculture^[1] or site-specific crop management. In 1993, the global positioning system (GPS) became fully operational with satellites providing 24 hr a day coverage of latitude, longitude, elevation, and time of day information. The GPS satellite technology has been of significant importance as it is considered a national asset that determines precise locations.^[2] Site-specific information is being used in geographic information systems (GIS) to layer information from different sources for advanced analyses to look for correlation of production factors and to search for limiting constraints. The entire intent of precision agriculture is to match inputs within a smaller area instead of treating a field as a whole unit. When precision technologies are used, a multidisciplinary process is involved that requires cooperation between the gathering and analysis of information and the decision process that prescribes an action or application.^[3] In pest management, the monitoring, diagnosis, and management response is usually action taken based on the detection of a pest or plant stress itself, which is a major strength of remote sensing technology.^[4] Precision technologies geared toward pest management are adopted by growers looking to lower production costs, improve quality, and increase crop yields. An expanding base of technology providers offer products made with high-resolution sensors and equipment designed to extract, analyze, and interpret visual information.

Remote Sensing of the Electromagnetic Spectrum

The electromagnetic spectrum represents a range of energy from photons of light that go from very short but high-frequency gamma rays, X-rays, and

ultraviolet rays to the visible portion of light that our human eyes see. The visible range covers the primary colors of blue, green, and red, and is utilized in both satellite and aerial imagery. The spectrum continues from the visible range into the infrared range, which is composed of near, mid, and far portions of energy. Remote sensing of vegetation is aimed at reflectance of energy in the near infrared portion and absorption of energy in the visible range. Color infrared imagery allows users to see into portions of the electromagnetic spectrum that is not visible to the human eye. Use of multiple remote sensing images across time (temporal resolution) has the benefit of detecting changes in plant health.^[5] A color-coded green vegetation index (GVI) image of a cantaloupe (*Cucumis melo* L.) field under stress from root-knot nematodes (*Meloidogyne incognita* Kofoid and White) can map the area of infestation and direct pest control efforts. Satellite data processed with an algorithm can isolate reflectance of vegetation from soil and water influences. The GVI images are used to identify and map areas that are underperforming in crop vigor. The images do not identify the cause of the stress but do alert field personnel on areas that need to be checked for ground truthing verification.

Satellites and Resolution

Satellite data can be accessed through the Internet with JPEG file format that is sent with 96 × 96 dpi resolution. Landsat imagery acquired at 30-m or greater pixels appears to offer limited use for site-specific management, as the number of pixels per acre is limited to less than 5. Landsat has six reflective bands used for spectral analysis.^[6] The French SPOT (Le Systeme Pour l'Observation de la Terra) satellites 2 and 4 have 10-m panchromatic (black and white) along with 20-m multispectral color imagery. SPOT 5, launched in May 2002, has 10-m resolution color.^[7] The newest commercial satellites with 4-m or less color pixels include IKONOS,^[8] QuickBird,^[9] and OrbView-3,^[10] all with 1-m or less panchromatic resolution. The binary digit technology that the new satellites are based on is 11-bit format. This allows for collection of data across a spread of 2048 integers (2 to the 11th power equals 2048). Landsat, SPOT, and Indian Remote Sensing^[11] satellites have 8-bit data, which is spread across a range of 256 integers.

Use of Satellite Data

Satellite imagery should not be expected to identify areas in a field initially infested with small insects such as whiteflies (*Bemisia argentifolli* Bellows and Perring) or melon aphids (*Aphis gossypii* Glover).

Imagery should also not be used for disease monitoring of virulent pathogens such as late blight [*Phytophthora infestans* (Mont.) de Bary] that need to be controlled with preventive fungicides in tomato (*Lycopersicon esculentum* Mill.) and potato (*Solanum tuberosum* L.) crops. But imagery can be used to map plant disease impacts on crops from pathogens such as *Fusarium* and *Verticillium*. Imagery can detect problem areas caused by established populations of pathogens, nematodes, insects, mites, and weeds. With the use of histograms that identify the pixel (picture element) count of digital data, the amount of area in each category can be determined.

Variable Rate Technology

High-resolution (5 m or less) or medium-resolution (5–20 m) satellite or aerial imagery can be used to create management zones that indicate variability of crop conditions in a field. A prescription can then be made based on these zones to vary the rate of pesticide applications. With advances in engineering of pesticide application equipment, variable rate technology (VRT) is now entering an advanced frontier. With annual upland cotton (*Gossypium hirsutum* L.) production grown on approximately 14 million acres in the United States, interest has been generated to bring precision agriculture technology into real-time, on-the-fly variable rate applications of PIX (mepiquat chloride), a plant growth regulator that controls excessive plant growth. The spin-off technology involved a transfer from the public sector into the private sector.^[12] This interest in VRT has also been applied to defoliation chemicals applied to reduce the foliage prior to cotton lint harvest.^[13]

CONCLUSION

Satellite imagery will allow information to be gathered across large areas of production agriculture. The crucial element in effectively using visual information is the turnaround time involved from imagery acquisition to processing and delivery to an end-user. With private industry bringing high-resolution imagery into the hands of pest managers, a change is occurring in how many crops are managed.

While satellite technology still needs ground truthing to verify field conditions, there is an incredible opportunity for an enhancement of the decision-making process involved with pest management. Numerous other developments will ultimately be discovered as the use of satellite imagery is only limited by the imagination of users.

REFERENCES

1. National Research Council. Precision agriculture in the 21st century: geospatial and information technologies in crop management. In *Committee on Assessing Crop Yield-Site-Specific Farming, Information Systems, and Research Opportunities*; National Academy Press: Washington, DC, 1997.
2. National Research Council. The global positioning system. A shared national asset. In *Committee on the Future of the Global Positioning System and Commission on Engineering and Technical Systems*; National Academy Press: Washington, DC, 1995.
3. Zilberman, D. Analyzing economic opportunities for emerging technologies. In *Workshop on Information Technology in Agriculture, Resources, and the Environment: Linking University Research and Commercial Application*; University of California: Berkeley, May 27, 2004.
4. LeBoeuf, J. Who are the end-users and what information do they really want (and are willing to pay for)? In *Proceedings of the Workshop on Remote Sensing for Agriculture in the 21st Century*; Oct 23, 1996. <http://www.cstars.ucdavis.edu/projects/ag-21/index.html> (accessed June 2004).
5. LeBoeuf, J. Practical applications of remote sensing technology—An industry perspective. *HortTechnology* **2000**, *10* (3), 475–480.
6. Short, N.M., Sr. Remote sensing and image interpretation and analysis. In *The Remote Sensing Tutorial—An Online Handbook*; Robinson, J., Ed.; 1997. Sponsored by Applied Information Sciences Branch, NASA Goddard Space Flight Center. (accessed October 2004).
7. *SPOT Image*. <http://www.spotimage.com> (accessed June 2004).
8. *Space Imaging*. <http://www.spaceimaging.com> (accessed June 2004).
9. *Digital Globe*. <http://www.digitalglobe.com> (accessed June 2004).
10. *OrbImage*. <http://www.orbimage.com> (accessed June 2004).
11. *Indian Space Research Organization*. <http://www.isro.org> (accessed June 2004).
12. Institute for Technology Development. In *Stennis Space Center*. <http://www.iftd.org> (accessed June 2004).
13. *In Time*. <http://www.govertime.com> (accessed June 2004).

Secondary Pest Resurgence

C. N. Rao

V. J. Shivankar

Department of Entomology, National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

Shyam Singh

Horticulture, National Research Centre for Citrus (ICAR), Nagpur, Maharashtra, India

INTRODUCTION

“Resurgence of insect pests” following the application of insecticides has been known for a long time. More than 50 species of insect and mite pests showed upsurge following insecticidal applications with diverse chemicals.^[1] In 1991, Bhanthal, Dhillon, and Dhaliwal^[2] listed 12 agricultural crops in which a resurgence of target or non-target insect pests was reported. The proliferation of broad-spectrum insecticides after 1949 has almost inevitably been followed by pest resistance, resurgence, and outbreak of secondary pests. Insecticides causing resurgence include some organochlorines, organophosphates (OPs), carbamates, and synthetic pyrethroids (SPs). No single class of insecticides has been identified to be free from resurgence induction.^[3] Pest outbreaks occur in different groups of insects after the application of a wide variety of insecticides with different modes of action. This has become a widespread phenomenon in the past few decades.

DEFINITION

Resurgence refers to an abnormal increase in a pest population or damage following insecticide application, often far exceeding the economic injury level or a situation in which a population, after having been suppressed, rebounds to numbers greater than before suppression occurred. It is a statistically significant increase in population density or damage by the target pest following insecticide application.^[4] These increases are called resurgence or “flare back” of the arthropod population. Replacement, also frequently referred to as a “secondary pest outbreak,” occurs when a major pest is suppressed and continues to be suppressed by a tactic, but is replaced by another pest, previously with minor status. In this instance, the primary pest is strongly affected by the tactic, but the secondary pest is not. Resurgence and replacement are ecological backlash phenomena observed in a number of agricultural systems and other situations.

CAUSES OF RESURGENCE

Various hypotheses have been proposed to explain the phenomenon of insecticide-induced resurgence. The precise mechanism responsible for resurgence varies with the type of insect, host plant, and various biotic and abiotic factors of the environment affecting the phenomenon. Although evidence has shown that resurgence is most often associated with conventional insecticides, the possibility of its occurrence exists with any management tactic. This is particularly true if the tactic is directly favorable to the physiology of the insect (for example, enhanced nutrition) or has an adverse effect on important natural enemies. Resurgence may occur because of a variety of factors related to the host plant, the insect pest, the non-target organisms in the agroecosystem, and the insecticide used (Fig. 1). Secondary pest outbreaks are believed to be partially a result of “hormoligosis,” e.g., sucking pests in cotton. Hormoligosis occurs when sublethal quantities of a stressful agent increase an organism’s sensitivity and response to environmental factors. In several cases, this response has increased reproduction. A new species may become a serious pest when its natural predators are killed; spider mites, for example, caused havoc when DDT and other insecticides killed their predators (Table 1).

PEST UPSETS

There could be three possible causes to explain pest upsets: 1) reduction of natural enemies by pesticides, along with the pest; 2) direct favorable influences of pesticides on physiology and behavior of arthropods; and 3) removal of competitive species.

Resurgence of *Heliothis* species after applications of monocrotophos and aldicarb was caused by increased ovipositional preference for treated plants. Resurgence in the brown plant hopper, *Nilaparvata lugens* (Stal.), following methyl parathion and decame-thrin treatments was also caused by increased plant

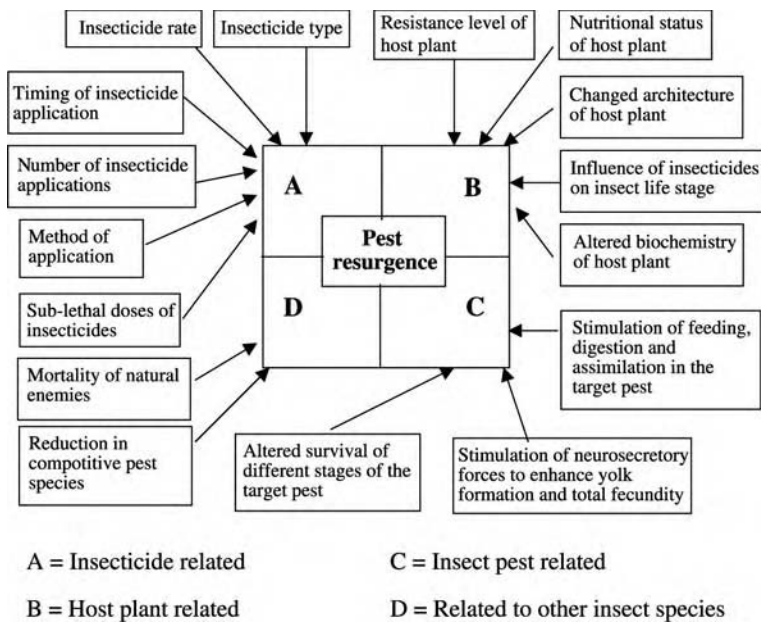


Fig. 1 Factors influencing pesticide-induced resurgence of insect pests. (From Ref.^[3].)

attractiveness (Table 1). If two or more species are competing in an area for the same requisite and one species is dominant, some workers believe that removal of the dominant will allow replacement by subordinate species, regardless of the changes in natural enemy populations. Most cases of resurgence occur in Homoptera (44%), followed by Lepidoptera (24%) and phytophagous mites (26%). It is interesting to note that some homopteran insects are protected from contact with insecticides by a waxy covering, and many of the lepidopterans exhibiting resurgence are borers and leaf miners, which also escape direct contact with the insecticides.^[1]

CLASSICAL EXAMPLES OF SECONDARY PEST OUTBREAKS

Apple

Prior to the introduction of DDT and OPs, the European red mite [*Panonychus ulmi* (Koch)] was not a pest on apples. After the advent of contact poisons, predators were nearly eliminated, and populations of European red mite increased unfettered; specific miticides had to be added to the apple spray program, at much greater expense to the growers.^[5]

Cotton

The use of SPs on cotton during the last 15 years has resulted in an increasing incidence of several sucking pests, including whiteflies, aphids, red mites, and mealybugs. Also, high applications of nitrogenous

fertilizers increase the tolerance of plant-sucking insect pests to SPs. Extensive resistance in *Helicoverpa armigera* (Hubner) to SPs (164- to 300-fold) was accompanied by secondary outbreaks of whitefly (*Bemisia tabaci* Gennadius) and spider mites on cotton in Andhra Pradesh, India.^[1] Appearance of bollworms, cabbage looper *Trichoplusia ni* (Hubner), beet army worm *Spodoptera exigua* (Hubner), tobacco caterpillar *Spodoptera litura* (Fabricius), aphids, spider mites, and whiteflies as serious pests of cotton in the United States is known to have been induced by the overuse of pesticides.^[6]

Rice

Most cases of insecticide-induced resurgence involved brown plant hopper [*N. lugens* (Stal.)] of rice from Bangladesh, India, Indonesia, and Solomon Islands (Table 1). The factors implicated include reduction in duration of nymphal stage, longer oviposition period, shortened life cycle, enhanced reproductive rate, higher feeding rate, and destruction of natural enemies especially *Cyrtorhinus lividipennis* Reuter.^[7]

Mustard

The application of SPs increased the concentration of glucose and some amino acids, especially arginine, lysine, isoleucine, leucine, and cystine, and caused resurgence of mustard aphid (*Lipaphis erysimi* Kaltentbach). These changes might be responsible for stimulation of reproduction and increased weight of such aphids. On the other hand, endosulfan did not

Table 1 Insecticides causing resurgence of important insect and mite pests of agricultural crops

| Insect pests | Crop | Insecticides |
|---|--------------------------------|---|
| I. Cereals | | |
| <i>Chilo suppressalis</i> (Walker) | Rice | HCH, parathion |
| <i>Cnaphalocrocis medinalis</i> (Guenee) | Rice | Carbofuran, phorate |
| <i>Empoasca maculifrons</i> (Motschulsky) | Rice | Acephate, demeton, dicrotophos, dimethoate, monocrotophos, phosphamidon |
| <i>Nephotettix virescens</i> (Distant) | Rice | Deltamethrin, phorate |
| <i>Nilaparvata lugens</i> (Stal.) | Rice | Acephate, azinophos methyl, BPMC, carbaryl, carbofuran, cypermethrin, deltamethrin, diazinon, ethoprop, fenthion, fenitrothion, fenvalerate, methomyl, miral, monocrotophos, parathion, permethrin, perthane, phorate, phosalone, phosphamidon, quinalphos, thiometon, triazophos, vamidothion, WL 8587 |
| <i>Sogatella furcifera</i> (Horvath) | Rice | Cypermethrin, deltamethrin, diazinon, fenvalerate, methyl parathion |
| <i>Rhopalosiphum maidis</i> (Fitch) | Corn | Fenvalerate |
| <i>Diabrotica virgifera</i> Le Conte | Corn | Carbaryl, carbofuran |
| II. Fruits and Vegetables | | |
| <i>Aphis malvae</i> (Koch) | Bitter gourd | Deltamethrin, malathion, permethrin |
| <i>Chaetosiphon tragaefolii</i> (Cockerell) | Strawberry | Disulfoton, phorate |
| <i>Leptinotarsa decemlineata</i> (Say) | Potato | DDT |
| <i>Myzus persicae</i> (Sulzer) | Brinjal | Cypermethrin, deltamethrin |
| <i>Polyphagotarsonemus latus</i> (Banks) | Chillies | Acephate, clocythrin cypermethrin, deltamethrin, fenvalerate, formathion, methyl demeton, monocrotophos, neem cake extract, permethrin, phosphamidon, thiometon |
| <i>Pseudococcus martinus</i> (Ehrhorn) | Grapevine | DDT |
| <i>Quadraspidiotus perniciosus</i> (Comstock) | Apple | Parathion |
| Phytophagous mites | Apple | Non-selective pesticides |
| <i>Tetranychus cinnabarinus</i> (Biosduval) | Muskmelon, watermelon, Brinjal | Fluvalinate |
| <i>Tetranychus urticae</i> Koch | Okra | Deltamethrin, fenvalerate Ethion |
| III. Commercial Crops | | |
| <i>Aphis gossypii</i> Glover | Cotton | Carbaryl, DDT, dimethoate, disulfoton, monocrotophos, phorate |
| <i>Bemisia tabaci</i> (Gennadius) | Cotton | Cypermethrin, deltamethrin, DDT, dimethoate, fenvalerate, monocrotophos, PP-321 |
| <i>Ferrisia virgata</i> (Cockerell) | Cotton | Cypermethrin, deltamethrin, fenvalerate, permethrin |
| <i>Tetranychus cinnabarinus</i> (Biosduval) | Cotton | Carbaryl, DDT, fenvalerate, fluvalinate, PP-321 |
| <i>Trialeurodes abutiloneus</i> (Haldeman) | Cotton | DDT, methyl parathion, toxaphene |
| <i>Bucculatrix thurberiella</i> Busck | Cotton | Carbaryl, synthetic pyrethroids |
| <i>Melanaspis glomerata</i> (Green) | Sugarcane | Endrin |
| IV. Oilseed Crop | | |
| <i>Lipaphis erysimi</i> (Kaltenbach) | Mustard | Cypermethrin, deltamethrin, endosulfan, fenvalerate, permethrin, phorate, phosphamidon |

(From Ref.^[3].)

affect the quality of leaf sap, but still caused enhanced reproduction and reduced the excretion of some amino acids in the honeydew of aphids, indicating that better utilization of these amino acids was responsible for the increased fecundity.

Sugarcane, Brinjal, and Chillies

The use of endrin to control borer, *Tryporhyza nivella* (Fab.), in sugarcane increased the population density of scale, *Melanaspis glomerata* Green. In brinjal, the

use of acephate against fruit borer, *Leucinodes orbonalis* Guenee, increased the population densities of mites and aphids. Similarly, the use of cypermethrin, fenvalerate, permethrin, and deltamethrin to control fruit borer in chillies increased the population densities of scale insects, mites, aphids, and wheat mites, respectively.^[8] Thus different insecticides may cause resurgence of the secondary insect pest by different mechanisms, and even the same insecticide may produce variable effects at different sublethal concentrations.

ROLE OF NATURAL ENEMIES AND INSECTICIDES

Like chemical control, parasitoids and predators may also disturb existing controls, resulting in pest resurgence and secondary pest outbreaks. In New Zealand, the introduced parasitoid, *Copidosoma floridanum* Ashmead, replaced native egg parasitoids, *Trichogrammatoidea* spp., as the principal natural enemies of the noctuid, *Chrysodeixis eriosoma* (Doubleday).^[9] Natural enemies can be suppressed by insecticides in a number of ways: 1) starvation because of host removal; 2) death from secondary poisoning after feeding on contaminated prey; 3) repellency of the insecticide; and 4) for internal parasitoids, if the host is killed before development is completed, death by starvation is assured. If the host survives treatment, the insecticidal effect may be mediated by the host's physiology. The host may detoxify the poison or possibly convert it to an even more toxic metabolite. Mortality from direct exposure to an insecticide is perhaps the most obvious means of natural enemy reduction. Insecticides detoxified oxidatively (several of the OPs) are much more toxic to natural enemies than to their phytophagous prey (hosts) because the former lack the detoxifying enzymes of the latter. Lacking the natural enemy check, secondary pests quickly multiply and attain key pest status.

MANAGEMENT OF RESURGENCE

The basic principle in dealing with resurgence and replacement is to combine several management tactics and thereby reduce the need for insecticide application. The fundamental objective is to avoid hormoligosis and the destruction of natural enemies in the agroecosystem. Another approach is to make inoculative releases of insectary-reared natural enemies after insecticide treatment residues have subsided and pest populations are recovering, to effect natural control once more. The newest, most unusual modification of the inoculative-release tactic is to release insecticide-resistant

natural enemies. The use of selective chemicals that have a stronger depressive influence on the pest population than on its natural enemies, e.g., microbial insecticides and insect growth regulators, and the use of otherwise non-selective chemicals in a selective manner help in maintaining the ecological balance.

CONCLUSION

“Resurgence of insect pests” following the application of insecticides has become a widespread phenomena observed in a number of agricultural systems in the past few decades. Resurgence refers to a significant increase in population density or damage by the target pest following insecticide application. “Secondary pest outbreak” occurs when a major pest is suppressed by a tactic, but is replaced by another pest with minor status. Resurgence may occur because of a variety of factors related to the host plant, the insect pest, the non-target organisms in the agroecosystem, and the insecticide used. Most cases of resurgence occur in Homoptera, followed by Lepidoptera and phytophagous mites. Insecticides causing resurgence of important insect and mite pests of various agricultural crops are listed in this article. Combining several management tactics including release of insecticide-resistant natural enemies, use of microbial insecticides and growth regulators, and use of non-selective chemicals in a selective manner and thereby reducing the need for insecticidal application holds the key in dealing with resurgence and secondary pest outbreaks.

REFERENCES

1. Jayaraj, S. Studies on the outbreak of whitefly, *Bernisia tabaci* (Gennadius) on cotton in Tamil Nadu. In *Resurgence of Sucking Pests*; Jayaraj, S., Ed.; Tamil Nadu Agricultural University: Coimbatore, India, 1986; 59–67.
2. Bhanthal, J.S.; Dhillon, G.S.; Dhaliwal, G.S. Insecticide induced resurgence in insect pests of agricultural crops. *Indian J. Entomol.* **1991**, *53* (3), 381–392.
3. Chelliah, S.; Bharathi, M. Insecticide-induced resurgence of insect pests of crop plants. In *Pesticides: Their Ecological Impact in Developing Countries*; Dhaliwal, G.S., Singh, B., Eds.; Commonwealth Publishers: New Delhi, India, 1993; 51–80.
4. Heinrichs, E.A.; Aquino, G.B.; Chelliah, S.; Valencia, S.L.; Reissing, W.H. Resurgence of *Nilaparvata lugens* (Stal.) populations as influenced by method and timing of insecticides applications in lowland rice. *Environ. Entomol.* **1982**, *11*, 78–84.
5. Horn, D.J. *Ecological Approach to Pest Management*; The Guilford Press, a Division of Guilford Publications, Inc.: New York, 1988; 285 pp.

6. Reynolds, H.T.; Adkisson, P.L.; Smith, R.F.; Frisbie, R.E. Cotton Insect Pest Management. In *Introduction to Insect Pest Management*; Metcalf, R.L., Luckmann, W.H., Eds.; John Wiley & Sons: New York, 1982; 375–434.
7. Chelliah, S. Insecticide-induced resurgence of rice brown plant hopper, *Nilaparvata lugens* (Stal). In *Resurgence of Sucking Pests*; Jayaraj, S., Ed.; Tamil Nadu Agriculture University: Coimbatore, India, 1987; 1–10.
8. Lal, O.P. Problems of insect pests and pesticide hazards. In *Recent Advances in Indian Entomology*; Lal, O.P., Ed.; APC Publications Pvt. Ltd.: New Delhi, 1996; 127–139.
9. Howarth, F.G. Environmental impacts of classical biological control. *Annu. Rev. Entomol.* **1991**, *36*, 485–509.

BIBLIOGRAPHY

Arora, R. Major insect pests of rapeseed-mustard. In *IPM System in Agriculture. Vol. V. Oilseeds*; Upadhyay, R.K., Mukherjee, K.G., Eds.; Aditya Publication (P) Ltd.: New Delhi, India, 1996; 45–51.

Sheep Predation

Eugene L. Fytche

Fytche Enterprises, Almonte, Ontario, Canada

INTRODUCTION

Sheep were among the first animals to be domesticated among nomadic peoples, providing wool, meat, and milk during their wandering. Their ability to graze on the limited vegetation in semi-arid regions was a valuable asset. Their vulnerability to attacks by wild animals was a problem as sheep had little capability of self-defense. Losses were inevitable, but because labor was cheap, their husbandry was based on full-time shepherds with dogs to defend their charges against predating wild animals. Sheep rearing is now universal.

In the following paragraphs, common predators and their killing techniques are described. Summary predation statistics is not readily available. Predation causes both economic and emotional trauma. Protective measures will be discussed in some details. Predators are products of Nature and their behavior is variable; as a result, protection may be less than 100% effective.

PREDATORS AND THEIR HABITS

Predators are bigger, stronger, fiercer, or smarter than the animals they prey on. In so far as sheep are concerned, wolves, dogs, coyotes, cougars, bears are common or occasional predators in temperate regions, jaguars in Central and South America, and a variety of other wild animals in Africa and Southeast Asia. Of these, domestic dogs are found wherever man lives, and feral dogs are common. Table 1 shows sheep population in selected countries, while Table 2 shows estimated losses in the United States of America in 1999. Ref.^[1] describes the ranges of wild predators, and Ref.^[2] their damage.

Wolves

Wolves occur in the Northern hemisphere, rarely in agricultural areas. In North America, their range is limited to northern Canada, with some exceptions. They are found in mountainous regions of Europe, Asia and Asia Minor, and North Africa. They generally operate in extended family groups that stay and hunt together. Their jaws are powerful enough to kill

by breaking a sheep's neck. The pack will consume the whole body of the prey, including the bones, each member having its place to feed in a rigid pecking order.

Coyotes

Coyotes, native to the North American prairies, have extended their range throughout the continent^[3] and are the major predator of American, Canadian, and Mexican sheep flocks, to the extent that many producers have been forced from the industry. The coyote can coexist with man. It is an opportunistic but skillful killer, may single out a sheep from the flock, and kill it without disturbing the rest of the flock. It normally seizes the throat of the sheep; its jaws are not strong enough to break the spine, so that the prey dies by asphyxiation. Some of the carcass will be eaten, and the family may return over several days. Carrion is acceptable food. Pups are trained to hunt within the family group and normally disperse in autumn, each to seek its own range.

Domestic and Feral Dogs

The domestic dog can become a killer, may operate singly, in twos, or in packs. While its killing sometimes patterns that of the coyote, it normally chases after frightened sheep, damaging the hind quarters of many of the flock. Because a domestic dog is rarely hungry, the carcass is never completely eaten. Trauma of the chase frequently causes pregnant ewes to abort. Feral dogs (frequently these are dogs turned loose by uncaring owners that learn to survive in the wild) may revert to instinctive killing in the same manner as wolves and coyotes. They kill to satisfy hunger but combine the feral instincts with an absence of fear of humans. They have become a major problem in Australia, where there are no wolves, coyotes, or large cats.

Cougars, Jaguars, and Bears

These three predators kill to satisfy hunger. Their size and fierceness make it difficult to protect against them.

Table 1 Sheep population in selected countries^a

| Country | Estimated sheep and lambs (million) |
|--------------------------------------|-------------------------------------|
| China | 130.0 |
| Australia | 118.6 |
| New Zealand | 39.5 |
| European Union (less United Kingdom) | 66.9 |
| United Kingdom | 29.7 |
| United States | 7.0 |
| Canada | 1.3 |

^aVarious sources, for year 2000.

They are strong enough to break the spine of a sheep, the first two while jumping on the sheep’s back, the last by blows of its powerful fore paws. Bears are less frequent killers of sheep, as an acceptable diet has more variety.

Other Predators

Foxes, lynx, bobcats, raccoons, eagles, ravens, vultures, and many other smaller predators will opportunistically attack lambs, rarely full-grown sheep. Their impact is small in total.

PROTECTIVE MEASURES

Protective measures^[4,5] can lessen the impact of predators on a sheep flock. Aspiring producers are well advised to do a “risk analysis” before investing time and money in a livestock venture. That would allow

Table 2 Predator kills, United States, 1999

| Predator | No. of head (1000s) | % of kills |
|------------|---------------------|------------|
| Coyotes | 165.8 | 60.7 |
| Dogs | 41.3 | 15.1 |
| Cougars | 15.6 | 5.7 |
| Bears | 7.8 | 2.9 |
| Foxes | 8.1 | 3.0 |
| Eagles | 10.7 | 3.9 |
| Bobcats | 12.7 | 4.7 |
| All others | 11.0 | 4.0 |
| Total | 273.0 | 100.0 |

Source: National Agricultural Statistical Service, USDA, May 5, 2000.

them to determine how, or whether, they should counter the risk. The conventional measures they might select include management practices, fencing, mobile protectors, and hunting and trapping. Research programs still investigate the effectiveness of other methods, some of which show promise. Full-time shepherds are rare, as a high proportion of sheep population is found in small farm flocks.

Management

Simple things can protect sheep in low-risk areas. Warning neighbors to keep their dogs at home, bringing the flock into barns at night, illuminating barnyards, pasturing close to the farmhouse, keeping sheep in pastures away from bush that might shelter predators, lambing in shelter, pasturing mothers and lambs close to the house, late afternoon feeding to condition the flock to return to the feeding station before dusk are some good and frequently effective practices.

Fencing

Good fences make good neighbors. Fences keep the sheep where the farmer wants them to graze. Few traditional fences^[5] will keep predators out: they can go over, under, or through log, woven wire, or other forms of fencing that are adequate to keep the sheep at home. High-voltage electric fencing,^[6-8] with a minimum of five suitably spaced wires alternating ground and live conductors, can be effective. Coyotes may solve this barrier over time, and in some locations a fence of 16 wires has been used to make the barrier “predator proof.”^[8] Protection against tunneling may be necessary. If good woven wire fencing exists, protection may be provided by energized outrigger wires. Electric fencing can be effective against wolves and bears, but its effectiveness against large cats is not established.

Table 3 Effectiveness of mobile protectors

| Mobile protector | Effectiveness ^a |
|------------------|----------------------------|
| Guard dog | 40–70% |
| Guard llama | 80% |
| Guard donkey | 50–60% |
| Owner | Variable |

^aRatings of effectiveness are subjective, by owners. Guard animals must be properly bonded, have territorial sense, and have aversion to canines to be effective. (From Refs.^[9-11])

Mobile Protectors (Guard Animals)

Mobile protectors are animals that either by bonding with the sheep, through an instinctive hate for canines, or through a strong territorial instinct, will drive off predators. Donkeys, llamas, and dogs exhibit these characteristics to a degree dependent on the individual animal. Effectiveness is summarized in Table 3. The owner may be considered as a mobile protector as well.

Donkeys^[9] can be bonded with sheep, preferably at an early age, by gradual introduction to the flock. They abhor canines and chase them from the pasture. Several donkeys in the same field do not improve protection, as the donkeys neglect the flock to enjoy the company of their own kind. A single donkey is vulnerable to packs of coyotes and wolves. “Standard” donkeys, preferably females, are recommended, as miniature donkeys are too small to deal with wolves and large coyotes. They are long lived and usually are not aggressive to humans.

Llamas^[10] will bond to the sheep flock and will chase predators from the pasture. The elevation of their eyes gives them a better oversight than donkeys, and it appears that several llamas can work together effectively. They have long lives and are not aggressive to humans. They are vulnerable to packs of canines.

Guard dogs^[11] have been used to guard flocks of sheep and goats since biblical times. It is essential that they be introduced early to the flock and allowed to mature in a program that ensures that they are not faced with situations that they are not mature enough to handle. They should not become pets and should not be overtrained as generally this will reduce their effectiveness to act independently as flock guardians. Several dogs will work together and are big enough to fight wolves and bears, but single dogs will have trouble with wolf packs. Because they are strong dogs, they must be carefully managed, and neighbors should know that they are not pets. Their average life span is less than either donkeys or llamas.

Other animals may have some protective value—horses and highland cattle, for example.

Hunting and Trapping

Hunting of wild predators is inefficient. Killing a predator that is not killing sheep may open its range for a predating animal. In the case of coyotes, it is generally accepted that intensive hunting leads to larger litters. A sheep producer can only hunt at the expense of other activities. Professional hunters, if available, may be expensive, and there is no guarantee that they will shoot the problem animal.

Trapping, including snaring, makes lesser demands of time, but is a skilled trade that may be done badly by a producer without expertise. Trappers also may

be expensive but are more successful in eliminating the problem animals.

Other Protective Measures

Experimentation with other methods has shown limited or temporary success. Various poisoning devices have been tried, targeting killers, and are more selective than the practice of poisoning carcasses. Sonic or supersonic noisemakers and visual deterrents may have temporary effectiveness. Taste aversion and smell deterrents have been tested with inconsistent results.

CONCLUSION

Domestic sheep are found all over the world except in the polar regions. Full-time shepherds are disappearing in developed countries, and more and more sheep are left with sporadic supervision. In all locations they are vulnerable to predators.

Many species of wild predators kill for food, but domestic dogs may kill for love of the chase. Methods of limiting their activity are many and must vary with the country and the species. Perhaps the universal remedies are hunting and trapping, but these are inefficient in use of time and are ineffective because the target, the problem animal, may escape.

Fencing can be effective in preventing entry of foxes, dogs, wolves, and coyotes, but may be ineffective for cougars, lynx, and other large cats. There is little protection for small lambs against flying predators, other than roofed enclosures. Guard dogs have a long history of successful protection, in both range and farm applications. They must be carefully managed, and average working life is short. Llamas and donkeys, if bonded properly with the flock, can be effective with farm flocks but may be less effective on open range. Experimentation with other methods continues but show little promise for universal application.

Producers, when faced with a predation problem, must make an evaluation of the risk and choose the protection that fits their specific case. They may find that cunning predators may solve the defense they have chosen, and they may have to change it, or double up. It is “what works for them” that counts.

REFERENCES

1. Burt, W.H.; Grossenheider, R.P. *A Field Guide to the Mammals of North America*; Houghton Mifflin Co.: Boston, 1976.
2. Wade, D.A.; Bowns, J.E. *Procedures for Evaluating Predation on Livestock and Wildlife*; Texas

- Agricultural Extension Service College Station: Texas, 1997.
3. Parker, G.R. *Eastern Coyote. The Story of Its Success*; Nimbus Publishing Ltd.: Halifax, Canada, 1995.
 4. Fytche, E.L. *May Safely Graze*; E.L. Fytche & Creative Bound International Inc.: Carp, Canada, 1998.
 5. Fytche, E.L. *Wild Predators? Not in My Backyard*; E.L. Fytche & Creative Bound International Inc.: Carp, Canada, 2003.
 6. Sheldon, M.; Gates, N.L. *Anti-Predator Fencing: A Synopsis of Research*; Agricultural Research Service, USDA: Beltsville, MD, 1987.
 7. *Gallagher Power Fencing Systems*; Manufacturer's Catalogue: Hamilton, New Zealand, 2001.
 8. Building an electric anti-predator fence. PNW **1983**, 225. Pacific Northwest Extension Publication.
 9. Walton, M.T.; Field, C.A. Use of donkeys to guard sheep and goats in texas. Proceedings of the Eastern Wildlife Damage Control Conference, 1989.
 10. Franklin, W.I. *Guard Llamas*; Iowa State University Extension, 1994. Doc. PM1527.
 11. USDA. Livestock guarding dogs: protecting sheep from predators. Agric. Inf. Bull. **1990**, 588.

Silverleaf Whitefly Management Using Reflective Plastic and Wheat Straw Mulch

Charles G. Summers

Department of Entomology, University of California, Parlier, California, U.S.A.

James J. Stapleton

Statewide IPM Project, UC Kearney Agricultural Center, Parlier, California, U.S.A.

Jeffrey P. Mitchell

Kearney Agricultural Center, University of California, Parlier, California, U.S.A.

INTRODUCTION

Ultraviolet (UV)-reflective plastic and wheat straw mulch were effective in repelling adult silverleaf whiteflies, *Bemisia argentifolii* Bellows and Perring, from zucchini squash, pumpkin, and cucumber. This resulted in a delay in colonization and reduction in the number of whitefly nymphs infesting these plants. Squash plants growing over both reflective plastic and wheat straw mulches produced significantly higher yields than did those growing over bare soil. Yields of pumpkins and cucumbers were higher in plants growing over reflective plastic than in those growing over bare soil.

OVERVIEW

Silverleaf whitefly is a serious pest of cucurbits including zucchini squash, pumpkins, and cucumbers. Chemical control has not been effective^[1] and there are few examples of effective biological control in vegetables.^[2] Silverleaf whitefly rapidly develops resistance to all classes of insecticides.^[3,4] UV-reflective plastic mulches have been used successfully to delay colonization by silverleaf whitefly and reduce the incidence of squash silverleaf.^[5,6] These mulches reflect short-wave UV light,^[7] which confuses and repels incoming adult whiteflies, thus reducing their incidence of alighting on plants.^[6] Whereas recent studies have shown that wheat straw mulch deters alate aphids from landing,^[8,9] similar information is unavailable for whiteflies.

RESPONSE OF WHITEFLIES TO REFLECTIVE PLASTIC MULCH

Studies were conducted on several cucurbits to determine the efficacy of reflective plastic and wheat straw mulch in managing silverleaf whitefly.

Pumpkin

Reflective plastic mulch, as a means of managing whiteflies, was compared to bare soil. Fewer whitefly adults were found on pumpkins growing over reflective mulch than on those growing over bare soil (Table 1). Fewer numbers of adults resulted in fewer nymphs (Table 1). Feeding by whitefly nymphs causes a condition known as “silverleaf” in a number of cucurbits including pumpkins.^[10,11] This condition results in a reduction in photosynthesis,^[12] which ultimately affects yield. This is clearly shown in terms of both yield per plant and the number of fruit per plant. Both were higher in plants growing over reflective mulch (Table 1). The reduction in the number of nymphs clearly affected the amount and the appearance of silverleaf, with plants growing over reflective mulch presenting lower levels of this malady (Table 1). Silverleaf whitefly feeding causes bleaching of the fruit and stems of a number of plants.^[10] Pumpkin is also affected by this feeding, and the fruit from plants growing in the control plots was several shades lighter orange than fruit from plants growing over the reflective mulch.

In a 20-acre commercial field, pumpkins in one-half of the field were grown over reflective plastic mulch, and in the other half, they were grown over bare soil. At harvest, 100% of the plants growing over bare soil presented symptoms of silverleaf, whereas <25% of those growing over the reflective mulch presented such symptoms. Yields from the portion of the field planted over reflective mulch averaged 33,686 pounds of fruit per acre (3020 fruit per acre) compared to 8189 pounds per acre (915 fruit per acre) from plants growing over bare soil.

Cucumber

Cucumber is an excellent host for silverleaf whitefly. Adult whiteflies were found in very high numbers on

Table 1 Number of adult, nymphal silverleaf whiteflies, percentage of plants presenting silverleaf symptoms, yield per plant and the number of fruit per plant in pumpkins

| Sample number | Treatment | |
|---|--------------------------|-------------------|
| | Reflective plastic mulch | Bare soil control |
| <i>No. adults per leaf</i> | | |
| 1 | 0.7a | 2.1b |
| 2 | 0.8a | 5.3b |
| 3 | 1.7a | 20.2b |
| 4 | 1.0a | 9.9b |
| <i>No. nymphs per 3.25 cm²</i> | | |
| 1 | 1.4a | 10.6b |
| 2 | 3.2a | 17.3b |
| 3 | 4.7a | 34.9b |
| <i>Percent silverleaf</i> | | |
| 1 | 0.0a | 0.0a |
| 2 | 0.0a | 38.0b |
| 3 | 0.1a | 68.0b |
| 4 | 13.0a | 100.0b |
| Pounds of fruit per plant | 8.8a | 2.4b |
| Mean no. fruit per plant | 1.3a | 0.6b |

Means followed by the same letter are not significantly different at $P = 0.05$; Fishers protected LSD.

the first true leaf in plants growing over bare soil, and populations continued to increase (Table 2). Adult whitefly number on plants growing over reflective mulch, however, remained several orders of magnitude below those growing over bare soil (Table 2). This was also reflected in nymphal populations (Table 2). By the

Table 2 Number of adult and nymphal silverleaf whiteflies and yield per plot in cucumbers

| Sample number | Treatment | |
|---|--------------------------|-------------------|
| | Reflective plastic mulch | Bare soil control |
| <i>No. adults per leaf</i> | | |
| 1 | 1.5a | 373b |
| 2 | 8.5a | 855b |
| 3 | 11.1a | 522b |
| 4 | 18.1a | 469b |
| 5 | 17.4 | 310b |
| <i>No. nymphs per 3.25 cm²</i> | | |
| 1 | 2.1a | 34.4b |
| 2 | 3.2a | 49.6b |
| 3 | 5.1a | 107.5b |
| 4 | 1.4a | 38.9b |
| Yield pounds per plot | 60.1a | 5.9b |

Means followed by the same letter are not significantly different at $P = 0.05$; Fishers protected LSD.

time the plants began to bear fruit, those growing over bare soil were so stunted by whitefly feeding (Fig. 1) that virtually no fruit were produced. Cucumbers do not exhibit silverleaf symptoms, so no determinations were made. In 15 harvests, fruit yield from plants growing over the reflective mulch was 10 times that from plants growing over bare soil (Table 2).

The effectiveness of reflective plastic mulch in managing silverleaf whitefly was evaluated in a 5-acre commercial cucumber field, one-half of which was planted over reflective plastic mulch and the other half over bare soil (Fig. 2). Whitefly infestations were heavy with immature counts on plants growing over bare soil averaging ca. 70 nymphs per 3.25 cm² compared to < 10 per 3.25 cm² on plants growing over reflective mulch. The field was harvested 10 times and yields from plants growing over reflective mulch averaged 61,953 pounds of fruit per acre, whereas plants growing over bare soil produced only 13,760 pounds per acre.

RESPONSE OF WHITEFLIES TO REFLECTIVE PLASTIC AND WHEAT STRAW MULCH

Squash

In addition to reflective mulch, we evaluated wheat straw mulch and imidacloprid, a systemic insecticide, for management of silverleaf whitefly in zucchini



Fig. 1 Cucumber plants growing over reflective plastic mulch (bottom) and bare soil (top). Stunting to plants growing over bare soil is caused by silverleaf whitefly feeding.



Fig. 2 Commercial cucumber field one-half of which was planted over reflective plastic mulch (foreground) and the other half over bare soil (background).

squash. Both the reflective plastic and the wheat straw mulch delayed the buildup of silverleaf whitefly nymphs (Fig. 3) and the onset of silverleaf (Fig. 4). Figure 5 shows the effectiveness of reflective plastic and straw mulch in reducing the incidence of squash silverleaf. In 2000, yield of marketable fruit in the plastic and straw mulch plots was approximately twice that from the imidacloprid plot. In 2001, yield from the straw mulch plots was twice that of the imidacloprid and plastic mulch plots. Yields from both mulched plots ranged from 3 to 12 times higher than those from the control plots. The mulches were more effective than a preplant application of imidacloprid in reducing the severity and incidence of silverleaf whitefly.

Spectral Reflectance

Spectral energy distributions from the mulches were determined at 2-nm intervals between the wavelengths of 300 and 700 nm. The reflective plastic mulch was superior to both straw and bare soil in reflecting UV wavelengths from 300 to 400 nm. The plastic reflected 86% of the total incoming UV radiation. The reflected

UV serves to repel adult whiteflies as well as aphids. Across the UV spectrum there was no difference between the straw mulch and the bare soil. The reflective plastic mulch reflected 94% of the incoming photosynthetically active radiation (PAR), 400 to 700 nm, whereas the straw mulch reflected 85%. Bare soil reflected only 41% of incoming PAR. Increased plant growth and fruit production were definitely linked to reduced levels of insect and disease pressure, but it is also likely that PAR reflected back into the canopy helped contribute to increased growth, development, and production.

CONCLUSION

UV-reflective plastic and wheat straw mulch work well in repelling adult silverleaf whiteflies. This, in turn, delays and reduces the buildup of whitefly nymphs and the incidence and severity of silverleaf. Reductions in whitefly density resulted in significant yield increases in pumpkin, cucumber, and zucchini squash. Reflective plastic mulch deters whiteflies by reflecting UV

Red-Sub

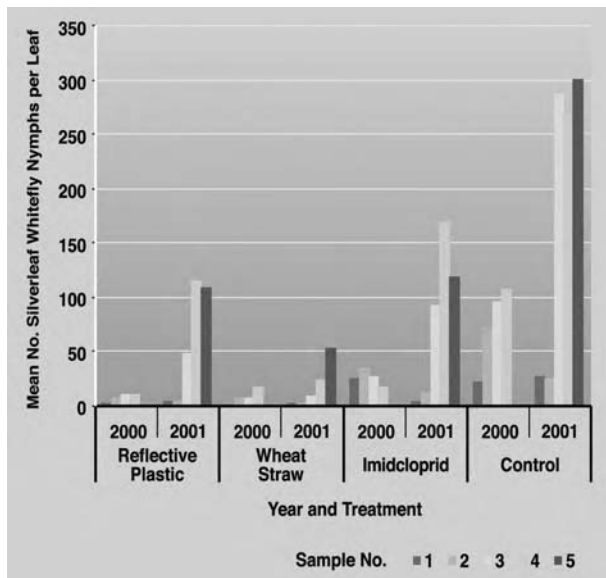


Fig. 3 Population density per leaf of silverleaf whitefly nymphs on zucchini squash leaves.

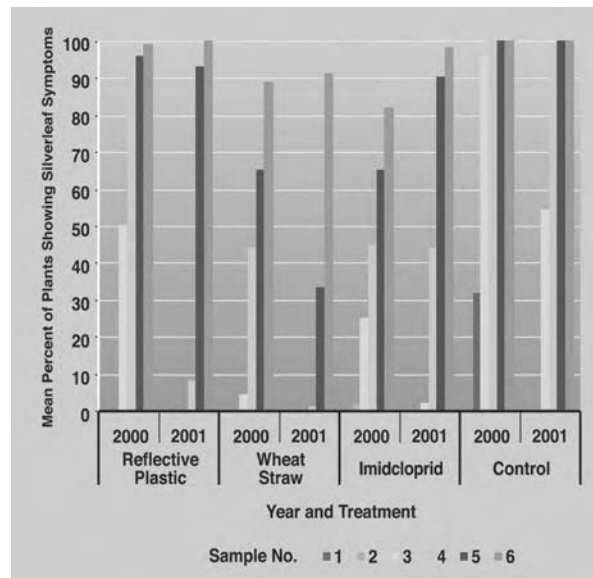


Fig. 4 Percentage of zucchini squash plants presenting squash silverleaf symptoms.



Fig. 5 Impact of reflective plastic and wheat straw mulch in reducing the incidence of squash silverleaf. First plot growing over wheat straw, second plot growing over bare soil, and third plot growing over reflective plastic. Note high incidence of silverleaf in second plot.

wavelength. This action is similar to that observed with aphids. The mechanisms behind adult whiteflies being repelled by wheat straw mulch is not known, because the reflectance of UV radiation from this medium did not differ from that of bare soil.

REFERENCES

- Horowitz, A.R.; Ishaaya, I. Chemical control of *Bemisia*, management and application. In *Bemisia: 1995 Taxonomy, Biology, Damage, Control and Management*; Gerling, D., Mayer, R.T., Eds.; Intercept: Andover, U.K., 1996; 537–556.
- Heinz, K.M. Predators and parasitoids as biological control agents of *Bemisia* in greenhouses. In *Bemisia: 1995. Taxonomy, Biology, Damage, Control and Management*; Gerling, D., Mayer, R.T., Eds.; Intercept: Andover, U.K., 1996; 435–449.
- Elbert, A.; Nauen, R. Resistance of *Bemisia tabaci* (Homoptera: Aleyrodidae) to insecticides in southern Spain with special reference to neonicotinoids. *Pest. Manag. Sci.* **2000**, *56*, 60–64.
- Prabhaker, N.; Toscano, N.C.; Henneberry, T.J. Evaluation of insecticide rotations and mixtures as resistance management strategies for *Bemisia argentifolii* (Homoptera: Aleyrodidae). *J. Econ. Entomol.* **1998**, *91*, 820–826.
- Edelstein, M.; Paris, S.H.; Shir, R.; Beibowitz, G.; Nerson, H.; Burger, Y. Effects of reflective plastic mulch on fall-grown summer squash. *Hassadeh* **1991**, *71*, 868–869.
- Summers, C.G.; Stapleton, J.J. Use of UV reflective mulch to delay the colonization and reduce the severity of *Bemisia argentifolii* (Homoptera: Aleyrodidae) infestations in cucurbits. *Crop Prot.* **2002**, *21*, 921–928.
- Harpaz, I. Nonpesticidal control of vector-borne viruses. In *Pathogens, Vectors and Plant Diseases: Approaches to Control*; Harris, K.F., Maramorosch, K., Eds.; Academic Press: New York, 1982; 1–21.
- Liewehr, D.J.; Cranshaw, W.E. Alate aphid traps capture over different background colors and different background patterns. *Southwest. Entomol.* **1991**, *16*, 13–18.
- Rummel, D.R.; Arnold, M.D.; Slosser, J.E.; Neece, K.D.; Pinchak, W.E. Cultural factors influencing the abundance of *Aphis gossypii* Glover in Texas high plains cotton. *Southwest. Entomol.* **1995**, *20*, 395–406.
- Costa, H.S.; Ullman, D.E.; Johnson, M.W.; Tabashnik, B.E. Squash silverleaf symptoms induced by immature, but not adult, *Bemisia tabaci*. *Phytopathology* **1993**, *83*, 763–766.
- Yokomi, R.K.; Jimenez, D.R.; Osborne, L.S.; Shapiro, J.P. Comparison of silverleaf whitefly-induced and chlormequat chloride-induced silvering in *Cucurbita pepo*. *Plant Dis.* **1995**, *79*, 950–955.
- Schmalstig, J.G.; McAuslane, H.J. Developmental anatomy of zucchini leaves with squash silverleaf disorder caused by the silverleaf whitefly. *J. Am. Soc. Hortic. Sci.* **2001**, *126*, 544–554.

Soil Infiltration by Pesticides

George Antonious

Water Quality Research, Kentucky State University, Frankfort, Kentucky, U.S.A.

INTRODUCTION

The extensive use of pesticides in agriculture has produced benefits that reduce pest infestations and crop loss, but also have various non-target impacts, such as the occurrence of pesticides in groundwater and surface water used for drinking water supplies. Pesticides have become an integral part of today's farming practices. Infiltration of pesticides from the land surface through soil and rock formations into groundwater and aquifers represents a potential human health problem. The watershed (the land area that drains into a river or stream) in which pesticides are applied can be the source of contamination of groundwater. Routine agricultural practices have resulted in the presence of 17 different pesticides in the groundwater of at least 23 different states. Some people believe that any amount of pesticides in their drinking water is unacceptable, whereas others feel that the standards and guidelines established for many of the major pesticides provide adequate protection. Which of these perspectives is closest to the truth remains unclear.

SOIL INFILTRATION BY PESTICIDES

Although considerable information is available on residues of pesticides in plants, soils, and water, relatively little is known about the levels at which such residues pose a human health hazard. Pesticides exert their toxic effects through repeated exposure and low-levels of daily exposure result in a "build-up process" that can cause overt illness. While pesticides provide many benefits, pesticides lost from the site of application no longer control pests and often have a detrimental effect on the environment. This is of particular concern in areas where residents use groundwater as a source of drinking water.

In the U.S., studies by the national water-quality assessment (NAWQA) program showed that pesticides are widespread in streams and groundwater within agricultural and urban areas. The most heavily used pesticides are found most often in geographic and seasonal patterns that mainly correspond to distribution of agricultural land and associated pesticide use. The perfect set of pesticide properties for preventing pesti-

cides from reaching groundwater would include a combination of low water solubility, tight binding to soil and rapid decay in the root zone. These properties would enhance low infiltration and rapid degradation rates. The root zone is the active surface zone of the soil profile where both biological and chemical processes, such as plant uptake, microbes degradation, and soil adsorption of pesticides, take place. This pathway of soil infiltration by pesticides could be an important factor to reduce pesticide impact on groundwater.

Many pesticides are tightly bound to the soil particles, degrade by soil microorganisms, and do not pose a threat to groundwater. On the contrary, the class of pesticides known as "nematicides" are highly soluble in water, have low tendency of soil binding, and are highly mobile in soil. These properties are required to control nematodes in soil deep in the root zone. Unfortunately, the same properties that make an efficient nematicide also increase the potential of groundwater contamination. Groundwater is the source of drinking water for more than 50% of the nation. It is common to think of surface water and groundwater as separate resources; however, they are interconnected. Groundwater discharge can significantly affect the quality and quantity of streams, especially during low-flow conditions. Likewise, surface water can affect the quality and quantity of groundwater.

In the last several years, there has been a major shift by U.S. farmers from plow tillage towards systems with reduced tillage. The reduced forms of tillage (conservation tillage) generally minimize soil erosion and water runoff, improve soil physical structure and productivity, and increase water infiltration rates into the vadose zone (the unsaturated water zone below the plant root). Concern has been raised regarding the environmental soundness of conservation tillage because of higher use of pesticides and generally greater rates of water infiltration, leading to leaching of pesticides to groundwater. Conservation tillage, therefore, may increase the potential for pesticide leaching. One consideration of the use of soil-applied pesticides is that they should persist long enough in soil to control the target pest(s), but not so long as to affect nontarget organisms or to create environmental risks.

Water infiltration through the soil profile provides the soil with the moisture content needed for seed

germination and seedling establishment. Infiltration may carry substantial amounts of pesticides (originating primarily from surface agricultural use) into the plant root area. Accordingly, soil infiltration by pesticides, especially insecticides and nematicides, has the advantage of controlling soil insects and nematodes. However, infiltration (seeping) of pesticides through the soil into the vadose zone, the layer that connects the point of chemical input and surface practices to the groundwater zone (the saturated water zone) must be reduced to ensure the safety of groundwater as a drinking water supply. Unfortunately, a low water infiltration rate accompanied by a heavy rain leads to flooding or runoff and erosion, particularly when agricultural operations occur on highly erodible lands. Factors that may influence the soil infiltration rate and pesticide transport through the soil profile include a series of physical, chemical, and biological properties of the soil as well as the properties of the pesticide under investigation as listed in Table 1. In addition to the properties listed in Table 1, subsurface drainage systems, if present, would also impact soil infiltration by pesticides. Accordingly, the distribution of pesticides throughout the soil profile, as a function of time, represents the integration of several processes such as mass flow, diffusion, adsorption/desorption, degradation, volatility, runoff, and plant uptake. One method of reducing the contamination of overland flow by soil chemicals (e.g., nutrients, pesticides) is to reduce their concentration at the immediate soil surface. This can be done by incorporating the chemical at lower depths, by either tilling the soil or by irrigation shortly after chemical application and/or development of pesticides that have high degradation rates on the soil surface.

Preferential flow or bypass flow is the non-uniform movement of water and transport of solutes from the topsoil into subsoil (undisturbed soil) through macropores (large non-capillary pores or channels) originated from soil cracks, old root channels, worm holes, etc., leading to rapid infiltration within and beyond the soil vadose zone. Preferential flow or macropores pathways allow water and solutes like fertilizers and pesticides to bypass the porous media of the soil matrix and get into groundwater. Pesticides and fertilizers are most susceptible to macropore transport, and thus should be the target of protective management practices. Short-circuiting to groundwater through macropores is a serious concern, because of the possibilities of rapid transport of toxic chemicals applied on the soil surface into groundwater.

The growing practice of minimum or no-tillage requires greater pesticide use on the soil surface with minimum incorporation into the soil, thus increasing soluble chemical amounts in overland flow that enters the macropores. In no-tillage, the plant residues on the

Table 1 Classification of some soil and non soil factors potentially affecting transport and soil infiltration by pesticides and other surface contaminants

| Soil factors | Non soil factors |
|--|----------------------------------|
| A—Soil physical composition | A—Climate |
| 1. Soil texture (% sand, silt, clay) | 1. Temperature and Humidity |
| 2. Soil organic matter content | 2. Rainfall intensity |
| B—Soil physical properties | B—Pesticide properties |
| 1. Bulk density | 1. Water solubility |
| 2. Field moisture capacity | 2. Volatility |
| 3. Hydraulic conductivity | 3. Soil adsorption coefficient |
| 4. Pore size distribution, macropores (natural openings), tendency to crack on drying. | 4. Ionization constant |
| | 5. Persistence in soil and water |
| C—Soil chemical properties | C—Pesticide application |
| 1. Cation- and anion-exchange capacity | 1. Foliar spraying |
| 2. pH | 2. Soil incorporation |
| 3. Redox potential | 3. Type of formulation |
| | 4. Rate of application |
| | 5. Time of application |
| D—Soil biological and biochemical properties | D—Agricultural practices |
| 1. Type and density of microorganisms | 1. Soil amendments |
| 2. Activity of soil enzymes | 2. Soil covers |
| | 3. Conservation tillage |
| | 4. Conventional tillage |
| | 5. Irrigation |

soil surface enhance worm activity and other macropore channels to stay open at the surface. The amounts of water and chemicals entering the macropore channels at the soil surface depend upon the amount of available overland flow (rainfall excess over infiltration), the amount of chemical that is transferred from the soil to overland flow, and the number and sizes of macropores. Accordingly, the transport through macropores will depend upon the soil type and porosity, soil surface condition, the degree of tillage, crop water uptake, and soil cover. The presence of a growing crop and preventing evaporation from the soil surface are good management practices, they increase infiltration and reduce macropore transport. Understanding the magnitude and the role of each of these factors on contaminant transport through macropores would guide us in selecting suitable management practices to minimize adverse impacts on groundwater.

Grass filter strips have become popular as a cover management practice to slow runoff and minimize soil erosion by trapping pesticides and runoff. The use of

herbicides to control weeds on erodible lands may reduce the need for tillage. However, when pesticide application coincides with intense thunderstorms or heavy rainfall, major surface runoff events may carry substantial amounts of pesticides causing soil and water contamination. According to a U.S. Environmental Protection Agency survey released in 1997, pollution is an epidemic problem in 21% of the nation's 2000 watersheds. Agriculture's part in this pollution problem is substantial. Management strategies to prevent surface water contamination must necessarily be based on a clear understanding of the processes that lead to contamination events. Some of the present management practices may create new problems while attempting to solve existing problems. The development of conservation tillage methods and their widespread implementation have demonstrated significant reduction of soil erosion, water runoff, and associated pollutants. This management practice also enhances and increases infiltration and recharge of aquifers.

Experiments conducted at Kentucky State University Research Farm (Franklin County, KY) using universal soil loss equation (USLE) standard plots established on a silty-loam soil (10% slope) described a reduction of runoff water, runoff sediment, and pesticides in runoff following natural rainfall when living fescue strips (Tall fescue, *Festuca elatior*, Kentucky 31, were planted across the contour of the slope as frequent barriers to runoff. Ten rows of buffer strips planted in pepper plots reduced Dacthal (DCPA, a herbicide) residues in runoff water and runoff sediment by 95% and 100%, respectively, compared to no-mulch (bare soil) treatment. This indicates that a substantial amount of Dacthal residues were trapped on sediment by the buffer strips along the hill slope that would otherwise have been transported down hill into surface water. When the number of buffer strips rows was

reduced to five rows per plot, Dacthal movement from the treated soil into runoff water and runoff sediment was reduced by 65% and 39%, respectively. Results also indicated that no-mulch plots planted with pepper intercropped with tomato as cover crop had 72% less runoff water and 79% less runoff sediment compared to no-mulch plots planted with pepper only. This is likely due to greater soil coverage in the mixed planting associated with the growth habit of tomato plants. Pepper has an erect growth habit and tomato has a prostrate, vining growth habit, resulting in greater soil coverage in the pepper/tomato plots.

Utilization of vegetative filter strips in agricultural fields resulted in a reduction of the transport of pesticides (e.g., endosulfan by 56%, Dacthal by 85%, clomazone by 81%) in runoff water allowing for their infiltration into the vadose zone. Increased water infiltration can result in the undesirable increased downward movement of pesticides. The mobility of any pesticide in soil is one of the principal parameters controlling the extent to which a pesticide may represent a risk for surface and groundwater contamination. Mulching has improved infiltration into the vadose zone as indicated by volume of water collected from the vadose zone (Table 2). Results indicated that cultivation of turf reduced runoff but did not reduce leaching of the water-soluble isomer of endosulfan (an insecticide) into the vadose zone. In spite of its low water solubility, Dacthal residues were detectable in the vadose zone indicating that its residues in soil are subject to subsurface flow. Water solubility is one of the pesticide characteristics that control infiltration and mobility. The movement of endosulfan and Dacthal from the soil surface into the vadose zone is a function of the availability of water as the transport agent (Table 2).

Once a pesticide enters the saturated zone its disappearance will be very slow. Important properties of

Table 2 Residue levels of Endosulfan and Dacthal (average of a 3-month sampling period) detected in water samples collected from the vadose zone following spraying under three soil management practices (Kentucky State University Research Farm, Franklin County, KY)

| Soil treatment | Pesticide | Infiltration rate, liter of water ha ⁻¹ | Pesticide infiltration, µg A.I. ha ⁻¹ of soil ^a |
|----------------|------------|--|---|
| Turf-1 | Endosulfan | 70–200 | 0.46–132.0 |
| | Dacthal | 29–311 | 70.8–758.8 |
| Turf-2 | Endosulfan | 46–179 | 12.9–50.1 |
| | Dacthal | 30–234 | 45.3–353.3 |
| No Mulch | Endosulfan | 25–162 | 3.5–22.7 |
| | Dacthal | 22–150 | 16.7–114.0 |

Turf-1: Living fescue strips, 30 cm wide, planted between every cropping row.

Turf-2: Living fescue strips planted every other cropping row.

No mulch: a bare soil.

^aSilty-loam soil of pH 6.7 (2% organic matter).

Adapted from Antonious, G.F. (1999) and Antonious and Byers (1997).

saturated zones are that they are dark, cold, and have less biological activity. The concentrations of the pesticides detected in the vadose zone will not, in most cases, be equivalent to concentrations in groundwater, because the pesticide will continue to decay as it traverses vertically to the water table and then horizontally to a well where it would be extracted. Once the pesticide reaches the groundwater, the rate of decay will be much slower and might remain in groundwater for a long period of time. To minimize the threat to groundwater contamination, pesticide applications should be timed to avoid periods of excessive rainfall.

FUTURE CONCERNS

Some pesticides were registered many years ago, when data requirements for registration were much less specific. Environmental transport and pesticide fate studies were not required until 1970. Nematicides, which are highly soluble in water, are currently the only effective means of reducing population densities of plant-parasitic nematodes in established orchards and vegetable crops. Further research is needed for all formulations of pesticides to clearly elucidate and modify the properties that control pesticide movement in soils.

BIBLIOGRAPHY

- Anonymous. *Agricultural Research*; U.S. Department of Agriculture, Agricultural Research Service: Greenbelt, MD, February 1998; 4–9.
- Antonious, G.F. Efficiency of grass buffer strips and cropping system on off-site Dacthal movement. *Bull. Environ. Contam. Toxicol.* **1999**, *63* (1), 25–32.
- Antonious, G.F. Clomazone residues in soil and runoff: measurement and mitigation. *Bull. Environ. Contam. Toxicol.* **2000**, *64* (2), 168–175.
- Antonious, G.F. *Pesticides in Runoff and Infiltration Water from Plant Production*, Kentucky Water Resources Annual Symposium, Feb, 23, 2001; Water Resources Institute, University of Kentucky: Lexington, KY, 2001; 47–48.
- Antonious, G.F.; Byers, M.E. Fate and movement of endosulfan under field conditions. *J. Environ. Toxicol. Chem.* **1997**, *64* (4), 644–649.
- Antonious, G.F.; Snyder, J.C.; Patel, G. Pyrethrins and piperonyl butoxide residues on potato and in soil under field conditions. *J. Environ. Sci. Health* **2001**, *36* (3), 261–271.
- Gjettermann, B.; Nielsen, K.L.; Patterson, C.T.; Jensen, H.E.; Hansen, S. Preferential flow in sandy loam soils as affected by irrigation intensity. *Soil Technol.* **1997**, *11*, 139–152.
- U.S. Geological Survey. The quality of our nation's waters—Nutrients and pesticides. U.S. Geol. Surv. Circ. no. 1225 **1999**, 82 pp.

Soil Management Practices

Randall C. Reeder

Food, Agricultural and Biological Engineering Department, The Ohio State University, Columbus, Ohio, U.S.A.

INTRODUCTION

Soil management practices have a direct effect on certain weeds, insects, and diseases that reduce crop growth and increase the cost of food production, but in comparing various tillage systems there is no single answer that applies to all pest management questions. Different species of pests often respond differently to changes in soil structure and surface environment.

Weeds, by definition, are undesirable. Insects, however, may be pest or beneficial. Many species of fungi, bacteria, viruses, and nematodes cause disease problems, but there are other species that do not.

Healthy plants are an excellent defense against pests. Stress opens the door for insects, disease, and weeds. Soil management practices that promote vigorous growth (early planting, rapid germination, and deep rooting) will lead to crops that can more likely tolerate pests.

improvements are short-lived if the same machinery is run over the soil again. Preventive measures for soil compaction include smaller loads, lighter axle loads, and lower tire pressures, but because of the need for bigger, more productive machinery, these are seldom adopted.

Controlled traffic can be a permanent solution for compaction problems. Machinery is adapted so all traffic runs in the same lanes and between rows. Typically, 20 to 30% of the field is trafficked, leaving the rest of the soil to be managed for ideal crop production, including pest management practices.^[2]

Soils that are too wet at critical times (planting and harvest) may be improved with surface and/or subsurface drainage. Timely field operations are important both for crop production and for pest management.

Precision placement of fertilizer, either under the row or beside the row, promotes rapid crop growth without fertilizing most of the weeds.

TILLAGE AND OTHER PRACTICES

Tillage and residue cover can be both a positive and negative influence depending on pest species.^[1] Whereas moldboard plowing and continuous no-till define the extremes of soil management on most cropland, a variety of tillage implements and attachments allow degrees of soil disturbance. Strip tillage is a modified form of no-till in which a narrow band of soil under the row is loosened, usually weeks or months before planting. To a potential pest, the area between the rows appears as no-till, whereas the row area has the characteristics of plowed ground.

Subsoilers typically till soil much deeper than a moldboard plow. Although they operate 35 to 50 cm deep, the resulting soil profile may be as loose and bare as a moldboard-plowed field or it may retain most of its structure and keep the surface fairly level and covered with residue.

Soil compaction, poor drainage, and low (or stratified) fertility levels are soil properties that tend to promote pest problems, indirectly if not directly. Late planting, poor germination, and shallow rooting lead to more weed pressure, e.g., weaker plants.

Subsoiling is often used to improve the soil structure in fields compacted by heavy machinery. The

WEEDS

Weed management is a challenge to anyone farming without tilling the soil. But if you think plowing is the solution to weed problems, remember we have been plowing for thousands of years and if plowing was a solution the weeds would all be dead by now. Eliminating or reducing tillage does change weed management strategies, and maybe calls for better management. Moreover, plowing (or timely cultivating between rows) can do a perfect job of killing existing weeds. No weed has ever been identified as “iron resistant,” but disturbing the soil also turns up more weed seeds, often creating an ideal environment for the next generation, and perennials can grow back from vegetative parts.

The amount of tillage to be done affects the choice of herbicide. Weed species in a given field will change in response to tillage system and herbicide program. Other factors can play a big role in weed management. Crop rotation, cover crops, and fertilizer amount, timing, and placement impact weed pressure and the difficulty of control.^[3]

High-quality soil will help seed germination and rapid, uniform crop establishment. A system that builds good soil structure and high organic matter can help the crop outgrow the weeds.

The influence of tillage and residue cover varies. Bailey and Goosen^[1] found that

Residue at the soil surface provides both pest and beneficial organisms with more favorable habitats for growth and survival. Wind-disseminated weed species (dandelion, foxtail barley, fleabane, narrow-leaved hawk's beard) and volunteer crops are commonly associated with reduced tillage systems, while invader species such as wild oat and millet that require soil disturbance to germinate and establish, are associated with conventional tillage. In contrast, other pests are indifferent to tillage system. Perennial weeds such as quackgrass, Canada thistle, and perennial sowthistle establish and spread in all tillage systems.

Conservation tillage systems are usually classified as mulch-till, no-till, or ridge-till. From a weed management standpoint, mulch-till is closest to moldboard plowing. The soil profile is loosened to about 10 cm or deeper. Although the soil is not inverted and much residue is left on the surface, the environment for weed seeds is often the same as for conventional tilled fields.

With no-till, previously buried weed seeds are not brought to the surface, and the crop residue left on the surface after planting also inhibits weed germination. However, perennial weeds that survived the previous season must be dealt with.

Ridge tillage provides benefits of both conventional and no-till. Ridge-clearing attachments on the planter move an inch or so of soil from the row area, along with weed seeds and seeds from the previous crop. Weeds that grow between rows can be controlled with one or two cultivations. This process makes ridge-till particularly effective against annual grasses. A band of herbicide applied over the row manages the few weeds that germinate on the ridge. The second cultivation helps control later weeds between the rows and rebuilds the ridges. The loose soil thrown up around the standing crop covers any small weeds in the row. A combination of cultivation and herbicides can provide excellent weed management.

Any system where the soil surface is not tilled every year or two may require better management to prevent some weeds from getting beyond control with common herbicides. Continuous no-till has many advantages and is worth the extra management effort that might be needed for selecting the right herbicides, rotating herbicides to avoid resistance, selecting varieties, rotating crops, and growing cover crops.

INSECTS

Pest management strategies are seldom changed for different tillage systems. Crop rotation and crop

history have a bigger influence on insects than soil properties. Rotating crops disrupts the life cycles of pests, and crop varieties can be selected to take advantage of genetic resistance to specific insects.^[4]

Whereas most research has focused on harmful insects, some research has looked at how soil management practices influence beneficial and non-target insects. Results are mixed, but in general, reducing disturbance of the top few inches of soil gives greater survival of ground beetles, rove beetles, spiders, and ants. This group of arthropods includes a number of generalist predators that feed on other insects.

Research with ground beetles, a large family of beetles that includes many predators, has given varied results from different tillage systems. Research in North Carolina and Georgia showed that no-till increased populations compared with conventional. Research in Ontario, Canada, and other states showed no influence. Conditions of the soil, crop, and residue cover that favor beneficial species also favor certain pests, so increases in the number of predators does not necessarily lead to fewer pests.^[5]

DISEASE

The effects of soil management practices on the development of crop diseases are variable. The largest numbers of diseases affected by conservation tillage systems are those whose pathogens survive in infected crop residue left on the surface. These diseases include foliar diseases, ear rots, and stalk or stem rots, and are the worst where continuous cropping is practiced. An example is gray leaf spot on continuous corn. Diseases favored by cool, wet soils may be more prevalent with no-till systems, whereas diseases favored by higher soil temperatures and drier soils may be less of a problem.

Pathogens include fungi, bacteria, viruses, and nematodes. Tillage system selection is often less of a factor for pathogens than are rainfall, relative humidity, air temperature, and soil type.^[6]

SLUGS AND RODENTS

Other pests affected by soil management practices include slugs and rodents. Although slug damage is not widespread geographically, they can decimate a crop. In one Ohio survey of slug damage to cornfields, 10% of conventional tilled fields had more than 20% stand injury, and 18% of no-till fields had the same level of injury. The buildup of residue in no-till fields increases the incidence of slug damage, but environmental factors are more critical. Slugs are most active in cool, wet conditions.^[7]

Crop residues in no-till fields provide an ideal habitat for several species of rodents that can easily grow in number and become major pests. Mice, voles, ground squirrels, and kangaroo rats are examples of pests that thrive in heavy residue, cover crops, and weedy conditions. Although tillage can help, the areas surrounding crop fields can contribute enough breeding stock to quickly repopulate a clean tilled field.^[8]

BENEFICIAL ORGANISMS

The amount and distribution of crop residue affects the populations of microorganisms that break down residues. Tillage distributes the microbial biomass through the soil profile and exposes the organisms to unfavorable environmental conditions. With no-till, microorganisms are concentrated near the surface. Fungal populations, e.g., become twice as large as with conventional tillage. Most fungi are beneficial, and under no-till the population of the “good” fungi increases up to 95%.^[11]

Clapperton^[9] found that “earthworms prefer plant material that has been colonized by fungi and bacteria, which can lead to the reduced incidence of fungal diseases in crops.” Earthworms thrive in undisturbed soil, and the benefits of the earthworm burrows can last for years if they are not destroyed by tillage. In no-till fields with up to 300 earthworms per square meter, there was a lower incidence of common root rot compared with conventional tilled fields where they found no earthworms.^[9]

BENEFICIAL SOIL MANAGEMENT PRACTICES

If soil management practices are neutral (neither positive or negative) on pest populations, then no-till or reduced tillage is much better overall. Soil erosion is typically 5 to 20 times greater from moldboard-plowed ground as from no-till, depending on the amount of crop residue. Residue cover is the single most important factor in reducing water erosion.^[10] Crops that produce little aboveground residue, such as cotton and soybeans, need to be followed with a cover crop to minimize erosion.

A protective cover of living vegetation or residue on the surface is also the surest and simplest way to reduce wind erosion. The relative soil loss reduction, compared with no cover, is about 70% for a 30% soil cover, and over 90% if at least 60% of the ground is covered.^[11]

On average, the United States is losing soil from cropland at about 10 t/ha/yr, a rate 10 times faster than top soil is being reformed.^[12] Worldwide, soil erosion averages 30 t/ha/yr on cropland, and about

80% of the world’s cropland is moderately or severely eroded.^[13,14]

Increased soil water conservation resulting from no-till practices, especially in subhumid and semiarid regions, often leads to higher crop yields. Surface residue results in less runoff and lower evaporation. In one Texas, USA, study, no-till stored almost 40% more soil-available water than moldboard plowing, and the resulting sorghum grain yield was 30% higher.^[15]

Soil carbon levels are directly related to tillage practices. The link between global warming and atmospheric carbon dioxide, a greenhouse gas, has increased interest in storing carbon, as soil organic matter, in agricultural soils. Reicosky^[16] measured cumulative carbon dioxide losses over 24 hr for various tillage tools and found a direct relationship to the volume of soil disturbed by the tillage tool. The moldboard plow lost 13 times more carbon compared to soil not tilled, and four conservation tillage tools averaged a loss of 4 times. The smaller carbon loss from conservation tillage tools shows a potential for carbon sequestration from no-till, strip-till, and other conservation tillage practices.

CONCLUSION

Soil management practices, along with choices about crops grown, rotations, cover crops, and fertility, have a major influence on problems associated with weeds, disease, and insects.

As no-till and other conservation tillage systems have grown in popularity, the lack of moldboard plowing is sometimes blamed, unfairly, for problems with pests. Although a decision on what tillage system to use may be influenced by pest management issues, there is no clear-cut, absolute answer. A farmer might change or give up a tillage practice because of a problem with a particular weed species, insect, or disease, but these pests will seldom be the dominant factor in selection of a tillage option. New technology (machinery designs, including planters and subsoilers), genetically modified crops, herbicides, and government policies have a greater impact on soil management decisions than pests.

REFERENCES

1. Bailey, K.L.; Goosen, B.D. Tillage. In *Encyclopedia of Pest Management*; Marcel Dekker: 2002; 837–839.
2. Reeder, R.C. Controlled Traffic. In *Encyclopedia of Soil Science*; Marcel Dekker, 2002; 233–236.
3. Martin, A.; Hayes, R.; Wicks, G. Weed Management. In *Conservation Tillage Systems and Management*.

- MWPS-45; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 120–138.
4. Rehm, G. Integrated crop management. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 102 pp.
 5. Royer, T.B.; Leonard, R.; Bagwell, R.; Leser, J.; Steffey, K.; Gray, M.; Weinzierl, R. Insect management. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 140–154.
 6. Jardine, D.; McMullen, M.P.; Sweets, L.E.; Kaufman, H.E. Disease management. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 156–166.
 7. Wilson, H.R. *Slugs on Ohio Field Crops*; 1992. <http://ohioline.osu.edu/icm-fact/fc-20.html> (accessed January 2004).
 8. Hines, R.; Hygnstrom, S. Rodent damage control. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 168–176.
 9. Clapperton, J. *The Real Dirt on No Tillage*; 2003 (accessed January 2004).
 10. Shelton, D.; Jasa, P.; Brown, L.; Hirschi, M. Water erosion. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 17–21.
 11. Lyon, D.; Smith, J.; Fryrear, D. Wind erosion. In *Conservation Tillage Systems and Management. MWPS-45*; MidWest Plan Service, Iowa State University: Ames, IA, 2000; 11–15.
 12. Unnevehr, L.J.; Lowe, F.M.; Pimentel, D.; Brooks, C.B.; Baldwin, R.L.; Beachy, R.N.; Chornesky, E.A.; Hiler, E.A.; Huffman, W.E.; King, L.J.; Kuzminski, L.N.; Lacy, W.B.; Lyon, T.L.; McNutt, K.; Ogren, W.L.; Reginato, R.; Suttie, J.W. *Frontiers in Agricultural Research: Food, Health, Environment, and Communities*; National Academies of Science: Washington, DC, 2003; 268 pp.
 13. Lal, R. Water management in various crop production systems related to soil tillage. *Soil Tillage Res.* **1994**, *30*, 169–185.
 14. Pimentel, D.; Harvey, C.; Resosudarmo, P.; Sinclair, K.; Kurtz, D.; McNair, M.; Crist, M.; Spritz, L.; Fitton, L.; Saffouri, R.; Blair, R. Environmental and economic costs of soil erosion and conservation benefits. *Science* **1995**, *267*, 1117–1123.
 15. Unger, P. No-till. In *Encyclopedia of Soil Science*; Marcel Dekker, 2002; 868–871.
 16. Reicosky, D.C. Tillage and gas exchange. In *Encyclopedia of Soil Science*; Marcel Dekker: 2002; 1333–1335.

Sorghum Insects: Ecology and Control

Gerald E. Wilde

Department of Entomology, Kansas State University, Manhattan, Kansas, U.S.A.

INTRODUCTION

Sorghum, *Sorghum bicolor* (L.) Moench, ranks fifth in hectareage and production among the grain crops of the world and constitutes a major source of food and feed in many countries. Worldwide, sorghum is grown on about 44 million ha.^[1] Sorghum is the third most important grain crop in the U.S.A., with 92% of production in seven states and 72% grown in two states, Kansas and Texas.^[2] Sorghum, thought to be originated in Africa, is now produced on six continents, in a zone extending about 40° on either side of the equator.^[3]

Sorghum is grown in different ways, as a subsistence crop in many areas of Africa and Asia and as a monoculture occupying large fields in the U.S.A. and Australia. More than 100 species of insects are considered pests of sorghum. Numerous reviews^[3-11] have discussed the importance of specific pests in different areas. The most recent thorough review of insect pests of sorghum by Teetes and Pendleton^[11] describes the biology, symptoms, and damage caused by sorghum insect pests, as well as techniques for monitoring and management. The purpose of this article is not to review what is already available but to provide insight into, and emphasis on, how the behavior and ecology of these pests and their natural enemies impact various facets of their management, including sampling, economic thresholds, and control tactics.

SORGHUM PEST MANAGEMENT

Yield losses caused by insect pests are estimated to cost US sorghum producers \$80 million annually.^[9] Worldwide, Judenko^[12] estimated losses during production to be 9%. Insect damage to stored grain has been estimated to be as much as 30% where grain is stored un-threshed in tropical environments.^[13] Several aphids are also important in transmission of viral diseases to sorghum. While many species of insects occupy the sorghum plant, as in many other crops, a relatively small number of key species cause the most damage on a regular basis. Key pests in the U.S.A. include the greenbug, *Schizaphis graminum* (Rondani); sorghum midge, *Stenodiplosis sorghicola* (Coquillett); and various caterpillars in southern areas. The major insect pests of sorghum on a global basis are the sorghum midge; shoot fly,

Atherigona soccata (Rondani); and several species of aphids, panicle bugs, and stalk borers.^[4] On a regional basis, various species of armyworms and locusts can cause catastrophic losses during outbreaks.^[7]

Management tactics include insecticides and natural enemies that cause mortality, or insect resistance and plant culture components that limit or slow growth of pest abundance. The role of the pest manager is to use knowledge about the ecology and behavior of the pest and its natural enemies to optimize strategies for monitoring and maintaining pest abundance below economic injury levels.^[14]

Identification

Several publications are available for identifying sorghum pest and beneficial insects in the U.S.A.^[15,16] The most comprehensive publication for identifying pest and beneficial arthropods in sorghum on a global basis is the Sorghum Insect Identification Handbook,^[17] which includes photos and descriptions of more than 50 species that inhabit sorghum.

Sampling

Timely scouting, using a variety of sampling techniques, is key to managing insect pests in sorghum. Because some pests attack plants at certain growth stages, knowledge of how a sorghum plant develops is essential.^[18] Early season pests occurring in or on the soil surface are estimated by visual examination of a specified area of the soil, soil surface, or seedlings. Visual examinations of plants are subsequently made, and the number of individuals per plant is recorded. For some pests that are difficult to detect or count, the number of severed plants, percentage of plants infested, or number of dead leaves are counted to estimate abundance. For chinch bugs, *Blissus leucopterus leucopterus* (Say), the number of insects in an alternate host (wheat) is used to predict the number of insects expected to infest sorghum. Panicle insects are usually assessed by visually counting the number per panicle. This practice is facilitated by shaking the head vigorously in a bucket so that even small larvae or nymphs can be detected. Various kinds of traps have been used to assess abundance of wireworms, shoot fly, and stalk borers, and sticky traps

Table 1 Economic thresholds for pests of sorghum

| Pest | Scientific name | Stage attacked | Threshold |
|--------------------------|--|----------------|---------------------------------|
| Wireworms | Family Elateridae | Seed, roots | 10/m ² , 2/trap |
| White grubs | <i>Phyllophaga</i> spp. | Seed, roots | 10/m ² |
| Red imported fire ant | <i>Solenopsis invicta</i> Buren | Seed | None |
| Cutworms | Family Noctuidae | Seedling | >5% cut plants |
| Shoot fly | <i>Atherigona soccata</i> (Rondani) | Vegetative | None established |
| Southern corn rootworm | <i>Diabrotica undecimpunctata howardi</i> Barber | Seedling roots | None established |
| Yellow sugarcane aphid | <i>Sipha flava</i> (Forbes) | Vegetative | 10–40% infested plants |
| Chinch bug—sorghum | <i>Blissus leucopterus leucopterus</i> (Say) | All stages | 2–3/seedling |
| Chinch bug—wheat | <i>Blissus leucopterus leucopterus</i> (Say) | Alternate host | 1/0.3 row meter |
| Greenbug | <i>Schizaphis graminum</i> (Rondani) | All stages | Varies with plant stage |
| Corn leaf aphid | <i>Rhopalosiphum maidis</i> (Fitch) | Whorl | Treatment not justified |
| Banks grass mite | <i>Oligonychus pratensis</i> (Banks) | All stages | More than 33% leaf area damaged |
| Corn earworm | <i>Helicoverpa zea</i> (Boddie) | Whorl, panicle | 1–2/head |
| Fall armyworm | <i>Spodoptera frugiperda</i> (J.E. Smith) | Whorl, panicle | 1–2/head |
| Sorghum webworm | <i>Nola sorghiella</i> Riley | Panicle | 2–4/head |
| Rice stink bug | <i>Oebalus pugnax</i> (Fabricius) | Panicle | 5/head |
| Southern green stink bug | <i>Nezara viridula</i> (L.) | Panicle | 4/head |
| Conchuela stink bug | <i>Chlorochroa ligata</i> (Say) | Panicle | 4/head |
| Leaf-footed plant bug | <i>Leptoglossus phyllopus</i> (L.) | Panicle | 6/head |
| False chinch bug | <i>Nysius raphanus</i> Howard | Panicle | 140/head |
| Sorghum midge | <i>Stenodiplosis sorghicola</i> (Coquillett) | Panicle | One adult/head |
| Stem borers | Family Pyralidae and Noctuidae | All stages | 3–8/trap |
| Grasshopper | Order Orthoptera | All stages | 15–20/m ² |
| Stored grain insects | Order Lepidoptera and Coleoptera | Storage | 2/kg |

have been used to monitor flights of chinch bugs and greenbugs.^[19] Stored grain pests are usually monitored with the use of a grain probe, and pheromone-baited sticky traps can be used to monitor stored grain moths.^[20] The sampling method for a specific technique in sorghum is similar to those used in most field crops. In general, representative areas of the field (four to five) are sampled, and the sample number is related to the amount of precision needed and cost per area sampled. In instances where border effects are known, such as with chinch bugs and sorghum midge, initial observations should be concentrated in those areas.

Economic Thresholds

Numerous studies have resulted in well-determined thresholds for some insect pests, but for most insect pests, thresholds are not well defined (Table 1).

The effect of an insect such as the greenbug and chinch bug varies, depending on plant growth stage; therefore, knowledge of plant development is needed

to adequately assess insect effects at a specific growth stage of the plant. A compilation of thresholds derived from various extension publications is presented in Refs.^[3,10,11] Thresholds may be expressed as numbers per unit area of soil or plant or the amount of visible plant damage. Recently developed computer models of sorghum plant growth and crop yield have great potential for better establishing relationships between damage by insect pests and yield loss. Such a model was used to establish the need for replanting resulting from damage by black cutworm.^[21] A decision support software program is available^[22] that interprets sampling data on stored grain insects and provides grain handlers with a risk analysis report detailing which bins are at risk for insect-caused economic losses.

Biological Control

Natural enemies (predators, parasitoids, and pathogens) play an important role in regulating insect pest abundance in sorghum. Little success has been

achieved with importation or augmentation of natural enemies, but conserving those occurring naturally in the sorghum agroecosystem has been very successful for managing pests. Although these natural enemies are important in regulating pest abundance, they do not always prevent pests from causing economic losses. Factors such as temperature, moisture, hyperparasites, and lack of alternate food limit the effectiveness of natural enemies. For example, sorghum midge abundance is not well reduced by parasites. Peters and Starkes^[10] listed 35 primary and 6 secondary parasites attacking sorghum pests as diverse as shoot fly, greenbug, stem borer, sorghum midge, and sorghum webworm. The presence and effectiveness of native predators in sorghum have often been overlooked in the past. Recent studies suggest that Coccinellidae and other predators play an important role in regulating aphid abundance in sorghum, and predators are important in reducing caterpillars that infest sorghum panicles.^[23] Several pathogens, usually fungi, affect abundance of some insect pests and may reduce abundance of chinch bugs; corn earworm, *H. zea* (Boddie); and fall armyworm, *S. frugiperda* (J.E. Smith). Several reports have suggested the use of nuclear polyhedrosis viruses to control *Heliothis* and *Helicoverpa* in sorghum. Conservation has most often been achieved by reducing or limiting the use of broad-spectrum insecticides in sorghum. For example, most outbreaks of Banks grass mite are associated with the effect of pesticides on mite predators. In recent years, the effect or role of landscape features (topography and plant species composition) on abundance of beneficial insects has been recognized. Landscapes may play an important role in the population dynamics of insect pests and natural enemies because many pests, especially aphids, also infest grasses and wheat, which are widely grown or occur in the same geographical zones in which sorghum is grown. Beneficial insects that occur in sorghum also contribute to natural enemy abundance in other crops, such as cotton, *Gossypium hirsutum* L., later in the season.

Cultural Management

Ecological features of the sorghum agroecosystem can lead to an increase or decrease in pest problems and, in many instances, can be manipulated for more effective management. Crop and alternate host destruction reduce food and overwintering habitats. A number of pests, including sorghum midge and sorghum webworm, *N. sorghiella* Riley, are reduced by these practices. Recent emphasis on reduced or no tillage to lessen erosion and moisture loss can reduce greenbug infestations on the one hand, and lead to an increase in pests such as cutworms and red imported fire ant,

S. invicta Buren, on the other. Planting date impacts insect pests such as the sorghum midge and other panicle-attacking pests, with early uniform planting resulting in less damage. Crop rotation is useful against pests with a limited host range, such as wireworms and white grubs, especially when sorghum is rotated with a broadleaf crop. Crop placement (planting sorghum away from wheat) is an effective management tool for chinch bugs in some areas because it prevents movement of chinch bug nymphs from wheat fields into adjacent seedling sorghum. Plant populations affect greenbug abundance in sorghum. Banks grass mite, *O. pratensis* (Banks), abundance in sorghum is usually associated with drought stress. When possible, adequate moisture through irrigation is encouraged.

Plant Resistance

Plant resistance has a long history in sorghum entomology as an effective management tool.^[24] The benefits from the development and deployment of insect-resistant sorghum include reduced crop protection costs and greater yields, as well as enhanced sustainability and conservation of natural and biological resources. Teetes^[25] listed 14 insect and 1 mite species for which resistant germplasm has been identified. Atlas sorgho, released in 1928, was used for many years as forage sorghum to control chinch bugs. Plant resistance essentially replaced insecticides for greenbug control in the 1970s. Use of plant resistance has been jeopardized somewhat by the greenbugs' ability to produce biotypes that attack formerly resistant genotypes. There is evidence that these biotypes are found on native grasses that occur in sorghum-growing areas of the U.S.A. Sorghum hybrids or varieties having open panicles are less infested by panicle-infesting species of insects. Efforts are continuing to detect and incorporate resistance into modern varieties and hybrids for several other key pests, including sorghum midge, shoot fly, Banks grass mite, and stem borers.^[4,6-8] Texas researchers^[26] estimated that a \$389 million annual net economic benefit to the US society resulted from greenbug-resistant sorghum developed in the 1970s. Development and use of molecular techniques could result in detailed characterization of individual loci implicated in sorghum resistance to insects.^[27]

Chemical Control

Insecticides offer the main defense against outbreaks or if other control options fail to keep pest abundance below economic thresholds. A variety of chemicals, usually applied as foliar sprays or granules, have been used against insect pests. Most insecticides are organophosphates, carbamates, or synthetic pyrethroids.

Recently, chloronicotinyl compounds having a different mode of action have been registered. These compounds have been used primarily as seed treatments and possess systemic activity, an advantage over the older organochlorine seed treatments, which do not. Chloronicotinyl compounds have seedling insecticidal activity against leaf- and stem-feeding insects such as greenbug and chinch bug as well as the seed-attacking pests such as wireworms and white grubs. They also possess insecticidal activity against common stored grain pests. The biology and behavior of some sorghum pests also affect the usefulness of insecticides. Greenbugs rapidly developed resistance to organophosphate insecticides in the 1980s when subjected to selection pressure. Sorghum midge populations often require multiple applications because new midges are emerging during an extended period of time. In Kansas, 11% and 9% of the sorghum hectareage were treated with a foliar-applied insecticide in 1991 and 1998, respectively.^[28] There are no published estimates of the percentage of hectareage receiving seed treatment with the chloronicotinyl compounds. In some areas of the U.S.A. where seedling pests such as the chinch bug occur, 50% of hectareage probably is treated.

CONCLUSIONS

The effective integration of control tactics for sorghum pest management is best served by knowledge of the behavior and ecology of the pest and its natural enemies, and their interaction with each other and the host plant. For example, there may be interguild and interspecific interactions between parasitoids and predators that may reduce their effectiveness. Plant resistance and parasitoids compliment each other in reducing greenbug abundance. Predators such as the minute pirate bug, *Orius tristicolor* (White), and *Coleomegilla maculata* (DeGeer) feed on plant tissues such as leaves or pollen, as well as their arthropod hosts; therefore, plant quality may impact their population dynamics. Density-dependent and -independent factors may also play a role in how natural enemies respond to pest abundance. Thus, the usefulness of some biological control agents depends on a pest's life history and the particular habitat occupied by the pest at a given time. In the future, better understanding of these kinds of interactions will lead to more sustainable pest management in the sorghum agroecosystem.

ACKNOWLEDGMENT

Contribution Number 05-335-B of the Kansas Agricultural Experiment Station.

REFERENCES

1. <http://apps.fao.org/> (accessed June 2005).
2. <http://www.usda.gov:81/1padb/grains.htm/> (accessed June 2005).
3. Young, W.R.; Teetes, G.L. Sorghum entomology. *Annu. Rev. Entomol.* **1977**, *22*, 193.
4. Seshu Reddy, K.V. Pest management in sorghum. In *Sorghum in the Eighties*; House, L.R., Mughogho, L.K., Peacock, J.M., Eds.; International Crops Research Institute for the Semiarid Tropics: Patancheru, A.P., India, 1982; 237–246.
5. Young, W.R. Sorghum insects. In *Sorghum Production and Utilization*; Wall, J.S., Ross, W.M., Eds.; Avi Publishing: Westport, CT, 1970; 235–287.
6. Jotwani, M.G.; Young, W.R. Recent developments in chemical control of insect pests of sorghum. In *Sorghum in the Seventies*; Rao, N.G.P., House, L.R., Eds.; Oxford and IBH Publishing Co.: 1980; 377–398.
7. Davies, J.C. Pest losses and control of damage on sorghum in developing countries, the realities and the myths. In *Sorghum in the Eighties*; House, L.R., Mughogho, L.K., Peacock, J.M., Eds.; International Crops Research Institute for the Semi-Arid Tropics: Patancheru, A.P., India, 1982; 215–223.
8. Jotwani, M.G.; Young, W.R.; Teetes, G.L. *Elements of Integrated Control of Sorghum Pests*; FAO Plant Production and Protection Paper; FAO: Rome, 1980; 159 pp.
9. Teetes, G.L. Sorghum insect pest management. In *Sorghum in the Eighties*; House, L.R., Mughogho, L.K., Peacock, J.M., Eds.; International Crops Research Institute for the Semiarid Tropics: Patancheru, A.P., India, 1982; 225–235.
10. Peters, D.C.; Starkes, K.J. Pest management systems for sorghum insects. In *Handbook of Pest Management in Agriculture*; Pimental, D., Ed.; CRC Press: Boca Raton, Florida, 1981; 549–562.
11. Teetes, G.L.; Pendleton, B.B. Insect pests of sorghum. In *Sorghum: Origin, History, Technology and Production*; Smith, C.W., Frederiksen, R.A., Eds.; John Wiley & Sons: New York, 2000; 443–495.
12. Judenko, E. Analytical method for assessing yield losses caused by pests of cereal crops with and without pesticides. In *Tropical Pest Bull.*, *2*; Center for Overseas Pest Research, Overseas Development Administration and Commonwealth Office, 1973.
13. Venkatarao, S.; Nuggehalli, R.N.; Swaminathan, M.; Pingale, S.V.; Subrahmanyam, V. Effect of insect infestation on stored grain. III. Studies on Kafir corn. *J. Sci. Food Agric.* **1958**, *(9)*, 837.
14. Pedigo, L.P. *Entomology and Pest Management*, 3rd Ed.; Prentice Hall: Upper Saddle River, New Jersey, 1999.
15. Norman, J.W.; Sparks, A.N., Jr.; Cronholm, G.B.; Jackson, J.A. *Field Guide to Pest and Beneficials in Texas Grain Sorghum*; Texas Agricultural Extension Service Pub. B-6094; 2000; 77 pp.
16. <http://www.oznet.ksu.edu/library/entml2/s121.pdf> (accessed July 2005).
17. Teetes, G.L.; Seshu Reddy, K.V.; Leuschner, K.; House, L.R. *Sorghum Insect Identification Handbook*;

- Information Bulletin No. 12; International Crops Research Institute for the Semi-Arid Tropics: Patancheru, A.P., India, 1982; 124 pp.
18. Vanderlip, R.L. *How a Sorghum Plant Develops*; Kansas Cooperative Extension Service Pub. S-3; 1993; 19 pp.
 19. Harvey, T.L.; Hackerott, H.L.; Martin, T.J. Dispersal of alate biotype C greenbugs in Kansas. *J. Econ. Entomol.* **1982**, 75 (1), 36–39.
 20. Hagstrum, D.W.; Flinn, P.W. Integrated pest management of stored grain insects. In *Storage of Cereal Grains and Their Products*; Sauer, D.B., Ed.; American Association of Cereal Chemists: St. Paul, Minnesota, 1992; 535–562.
 21. Heiniger, R.W.; Vanderlip, R.L.; Williams, J.R.; Welsh, S.W. Developing guidelines for replanting grain sorghum: III. Using a plant growth model to determine replanting options. *Agron. J.* **1997**, 89, 93–100.
 22. <http://bru.gmpcr.ksu.edu/proj/sga/> (accessed June 2005).
 23. Rice, M.E.; Wilde, G.E. Experimental evaluation of predators and parasitoids in suppressing greenbugs in sorghum and wheat. *Environ. Entomol.* **1988**, 836–841.
 24. Teetes, G.L. Breeding sorghum resistant to insects. In *Breeding Plants Resistant to Insects*; Maxwell, F., Jennings, P., Eds.; John Wiley Publishing Company: New York, 1980; 457–485.
 25. Teetes, G.L. Overview of pest management and host plant resistance in U.S. sorghum. In *Biology and Breeding for Resistance to Arthropods and Pathogens in Agricultural Plants*; Harris, M.K., Ed.; Texas Agricultural Experiment Station Pub. MP-1451: College Station, TX, 1979; 181–223.
 26. Eddelman, B.R.; Chang, C.C.; McCarl, B.A. Economic benefits from grain sorghum variety improvement in the United States. In *Economic, Environmental, and Social Benefits of Resistance in Field Crops*; Wiseman, B.R., Webster, J.A., Eds.; Proceedings Thomas Say Publications in Entomology, Entomological Society of America: Lanham, MD, 1999; 17–44.
 27. Nagaraj, N.; Reese, J.C.; Tuinstra, M.B.; Smith, C.M.; St. Amand, P.; Kirkham, M.B.; Kofoid, K.D.; Campbell, L.R.; Wilde, G.E. Molecular mapping of sorghum genes expressing tolerance to damage by greenbug (Homoptera: Aphididae). *J. Econ. Entomol.* **2005**, 98 (2), 595–602.
 28. Cress, D.C. *Kansas Agricultural Chemical Usage: 1998 Wheat and Sorghum Pesticide Summary*; Kansas Agricultural Experiment Station and Cooperative Extension Service Pub. MF-2437, 2000; 63 pp.

Soybean Diseases: Ecology and Control

Glen L. Hartman

Agricultural Research Service, United States Department of Agriculture
and University of Illinois, Urbana, Illinois, U.S.A.

INTRODUCTION

Soybean, *Glycine max*, is the domesticated member of more than 16 known *Glycine* species. Farmers in the eastern half of northern China domesticated soybean during the Shan dynasty (1550–1027 B.C.) or perhaps earlier, suggesting eastern Asia as its origin. For several thousand years, people in eastern Asia have used soybean seeds for animal feed, food, and as a medicine to treat a number of human disorders. Soybean was first introduced into the United States in 1765 and became widely planted and a major crop less than 150 years later.

As the growing area for soybean production has increased, the number and severity of diseases have also increased. Some diseases have been described recently, while others have been known for over 100 years. The economic importance of any one disease may vary from one geographic area to another and from one season to the next. The extent of loss depends upon the pathogen, environmental conditions, and the susceptibility of the soybean variety. Many pathogens can initiate an epidemic only under specific conditions. For example, many pathogens need certain moisture levels to infect soybean and others require certain vectors in order to be transmitted to plants. There are more than 200 pathogens or strains of pathogens known to affect soybean, about 50 of these are important economically. All plant parts are susceptible to pathogens, including roots, stems, petioles, leaves, flowers and seed.

BACTERIAL DISEASES

Bacteria are prokaryotic organisms found in air, soil, water, and on or in all plants and animals, including humans. Most lack chlorophyll, are saprophytic, have a rigid cell wall, and divide by binary fission. In a warm, moist environment, large numbers of new cells can be produced within a few hours.

Soybean bacterial pathogens multiply rapidly inside plants, where they cause death of cells (necrosis), abnormal growth (tumors), blockage of water-conducting tissue (wilting), or breakdown of tissue structure (dry

and soft rots). They often produce enzymes or toxins that induce yellowing (chlorosis), water soaking, and other symptoms like blight and leaf spots. The toxins or enzymes may migrate throughout the plant, causing systemic damage. Many infect or infest seeds and remain viable in or on them for one or more seasons, usually embedded in dried exudate produced while actively growing on soybean tissue. They are identified based upon their cell and colony morphology, host specificity, reaction to serological and biochemical tests, and the symptoms they cause. Common genera of plant-pathogenic bacteria that affect soybean include *Agrobacterium*, *Bacillus*, *Clavibacter*, *Curtobacterium*, *Erwinia*, *Pseudomonas*, *Rhodococcus*, and *Xanthomonas*. Most are unicellular rods up to 3 μm long, do not form spores (*Bacillus* spp. are an exception), and have one to several flagella. One genus, *Phytoplasma*, does not conform with the other plant-pathogenic genera, because cell walls are lacking and cell sizes are smaller (0.3–0.5 μm), and they are transmitted by leafhoppers.

Animals, flowing or splashing water, infected plant parts, soil, and wind-blown rain disseminates soybean bacterial pathogens. They can live epiphytically on soybean leaf surfaces and enter leaves through wounds or through natural openings. Water-soaked tissues often increase the susceptibility of plants to invasion by bacteria. Free moisture and moderate to high temperatures generally are required for pathogen and disease development.

FUNGAL DISEASES

Of the nearly 70,000 described species of fungi, more than 8000 are known plant pathogens. Fungi lack chlorophyll and cannot carry out photosynthesis, and instead function either as saprophytes or as parasites. Fungi are adapted for survival in air, soil, and water, and most produce microscopic, cellular, threadlike filaments called hyphae. Typically, fungi reproduce and spread by means of asexual or sexually produced spores that are dispersed by air currents, splashing or flowing water, and the activities of animals. Fungal taxonomy is based largely on the morphology of spores, the structures that produce them, and their development, along with molecular typing.

Soybean fungal pathogens cause a wide range of symptoms from root rots, wilt, stem lesions, leaf spots, pod and seed discoloration, and decay. Some species of *Cercospora*, *Colletotrichum*, *Diaporthe* (*Phomopsis*), *Fusarium*, and *Macrophomina* are common in soybean and can cause disease symptoms soon after infection or can cause latent infections.

Most fungal pathogens enter soybean plants indirectly through natural openings, such as stomata, hydathodes, nectaries, and lenticels, or they infect plants through wounds. A few of the fungal pathogens, like *Phakopsora pachyrhizi* (the cause of soybean rust) produce specialized structures such as penetration pegs that directly penetrate plant tissue through a combination of enzyme action and pressure. Many of the soybean pathogens, when not living in soybean tissue, exist on or in dead plants, seeds, soil, and occasionally insects. Some fungi, in addition to infecting soybean plants, infect and reproduce in weeds or other hosts.

NEMATODE DISEASES

Nematodes are roundworms lacking segments that inhabit animals, decaying organic matter, fresh and salt water, plants and soil. They are probably the most numerous multicellular animals on earth, and an estimated 7.5 billion nematodes live within the top 20 cm of a hectare of typical soil. Most of the more than 15,000 described species are microscopic, transparent, vermiform, mobile, and are not plant pathogens. Active movement of nematodes in soil is limited to less than 75 cm per year, but they passively move in water, soil, and infected plant parts. Nematodes are identified by the shape of various anatomical parts. The life cycles of most plant-parasitic nematodes include juveniles that hatch from eggs deposited by females in the soil or in root tissue, molt through four stages, and reach maturity after the final molt. Under optimal conditions, most plant-parasitic species complete their life cycles in three to four weeks.

Nematodes that infect soybean include more than 100 species that feed on, or are associated in some way, with the soybean roots, but only a few are of economic importance. Those species within the genera *Heterodera* (including soybean cyst nematode) and *Meloidogyne* (root-knot nematode) that penetrate and reproduce in soybean roots are most important. All nematodes that attack soybean are obligate parasites and must feed on living plants to complete their life cycle. Almost all feed on plant cells by puncturing cell walls with a hollow stylet, injecting secretory products into the cells, and ingesting the partially digested contents. Symptoms often mimic those induced by low or unbalanced fertility, poor drainage, drought, soil insects, root-rot fungi, or herbicides. Accurate diagnosis

of nematode problems requires analysis by a nematologist, including proper sample collection and handling of soil and roots.

VIRAL DISEASES

Viruses do not have a cellular form and live only within certain living cells as obligate parasites as macromolecular particles composed of genetic information in the form of nucleic acid that is either ribonucleic acid (RNA) or deoxyribonucleic acid (DNA) surrounded by a protective protein or lipoprotein coat. Virus particles can be seen only in a transmission electron microscope, with each virus having a specific size and shape. Most are isometric particles (roughly spherical) or elongated rods (either rigid or flexuous). Identifying a particular virus as the causal agent of a particular disease is a challenging job. Specific immunological and nucleic acid probes have enhanced the accuracy and speed of virus disease diagnosis. Viruses with common characteristics are grouped together into families, genera and species based on the nature, organization, and sequence of the nucleic acid genome; particle morphology; biochemical, physical, and serological properties; type of vector, if any; and host.

Symptoms caused by viruses on soybean include plant stunting, and yellowing or reddening of foliage that often becomes mottled. Identifying virus and virus-like diseases in the field can be difficult because symptoms on a given plant may be quite similar though induced by different viruses or by mixed infection with more than one virus. Some of the most common viruses on soybean include *Bean pod mottle virus* and *Soybean mosaic virus*. Soybean viruses exist in the cells of their hosts, and are transmitted to other hosts through wounds created by arthropod or nematode vectors, mechanical inoculation, pollination, and human activities including planting seeds harvested from virus-infected plants.

SOYBEAN DISEASE CONTROL

Soybean disease control or disease management includes preventative, preemptive and remedial strategies. Management of soybean diseases rarely can be accomplished in the long term by only one method. Disease management must be placed within the context of agronomic practices, state and federal land and pesticide use regulations, and economics. Integrated pest management, which utilizes economic injury levels, economic thresholds, scouting, record keeping, and mapping, allows for a planned, economically sound approach to disease management.

Preventive measures include the use of cultural management methods like crop rotation and planting of resistant or tolerant cultivars. Cultural practices include maintaining adequate growth conditions, with appropriate nutrients, water, plant density, and planting high-quality seeds in a favorable seedbed at the proper time. Effective cultural practices can reduce plant stress, and healthy, vigorous plants often suffer less yield loss from diseases than plants already under stress. Crop rotation is often effective because many soybean pathogens either die out or their populations are reduced when barley, maize, oats, rye, sorghum, or wheat are planted, since these are not hosts for most soybean pathogens. Planting resistant cultivars or varieties is of importance for a number of diseases including brown stem rot, downy mildew, frogeye leaf spot, *Phytophthora* root rot, soybean cyst, stem canker, and soybean mosaic.

Preemptive measures are adopted in certain areas where there is a repeated history of disease epidemics. Fungicides, insecticides, and nematicides are applied according to a certain calendar date or growth stage, and applications are made when damage is expected, regardless of what levels of injury are detected. For fungicides, both protective and curative fungicides have been registered for use. Correct applications with

minimal drift loss and thorough canopy penetration are critical to the success of foliar fungicide programs on soybean. Insecticides and nematicides are rarely used in soybean for ecological and economic reasons.

Remedial measures are adopted when disease occurrence reaches an established threshold or an economic injury level. There are few, if any, successful remedial activities that can be used for management of soybean diseases. Crop rotation, plowing and/or tillage practices can be used to eradicate or reduce the pathogen population. Some fungicides have localized eradication properties, and these may be of more use in controlling some soybean diseases, like soybean rust.

BIBLIOGRAPHY

- Boerma, H.R., Specht, J.E., Eds.; *Improvement, Production, and Uses*; American Society of Agronomy, Inc., Crop Sciences Society of America, Inc., and Soil Science Society of America, Inc.: Madison, WI, 2004; 1144 pp.
- Copping, L.G., Green, M.B., Rees, R.T., Eds.; *Pest Management in Soybean*; Elsevier Applied Science: London, 1992; 369 pp.
- Hartman, G.L., Sinclair, J.B., Rupe, J.C., Eds.; *Compendium of Soybean Diseases*; APS Press, Inc.: St. Paul, MN, 1999; 99 pp.

Strategies for Reducing Risks with Agricultural Pesticides in Developing Countries

Sylvia I. Karlsson

Finland Futures Research Centre, Turku School of Economics and Business Administration,
Tampere, Finland

INTRODUCTION

The negative effects on health and environment from agricultural pesticide use in developing countries are significant even if it is very difficult to give quantitative figures owing to lacking data. Developing countries account for a third of the world's pesticide market. Pesticides are primarily used in commercial agriculture but increasingly also in subsistence farming, and the conditions of use are often significantly increasing the health and environmental risks.

The major strategies that underlie current efforts to reduce risks from the use of pesticides in developing countries are based on various assumptions about causality, culpability, and responsibility. Each strategy is also supported by different types of institutions at different governance levels, and this has implications for the resources required for the different strategies.

THE PESTICIDE RISK FUNCTION

The causes behind health and environmental risks from the use of pesticides can be summarized as a function of three factors

$$\text{risk} = f\{\text{use(quantity)} \\ \times \text{type(category and quality)} \times \text{mode}\}$$

The concept of "risk" in this function denotes the probability of a particular adverse effect.^a Risks from chemicals for biological organisms are a function of the toxicity and exposure. In the function above, the toxic/ecotoxic characteristics are included in the type factor as is the quality of the pesticide product, the formulation. Exposure is a function of the quantity of pesticide used and the mode in which it is used. There are a number of circumstances related specially, but not only, to exposure patterns that make the health

^aThis is the common definition in relation to negative outcomes on biological organisms owing to exposure to a chemical agent.^[1] The opposing term to risk is in this context "safety," which is defined by individuals or governments as "a circumstance characterized by an 'acceptable' level of risk" (1:985).

and environmental risks different and often particularly high in developing countries.^[2-4] This calls for a closer look at the options for reducing risks.

RISK REDUCTION STRATEGIES

The efforts of reducing risks with pesticides in developing countries include a wide range of measures at several governance levels. The measures can be classified into three major risk reduction strategies that each address one of the three factors in the risk equation. The strategies focus on reducing the *use* of pesticides, targeting the worst *types* of pesticides, or improving the *mode* of using them. Naturally, many risk reduction measures address more than one of these factors.

Reducing the Use of All Pesticides

The strategy to reduce the use volume of pesticides includes measures encouraging organic farming and integrated pest management (IPM). Organic farming excludes the use of all chemical pesticides. Many forms of IPM encourage the reduction of the overall volume of pesticide applications. Research and implementation on classical IPM concepts such as biological enemies, timing of spraying, and counting pests to determine economic threshold levels have existed for decades. A more ambitious form of IPM taught through Farmer Field Schools has been successfully applied in several Asian countries and is now spreading to other continents.^b In recent years, the strategy to reduce the overall use of pesticides has increasingly been pushed by intergovernmental organizations (IGOs) through their stress on IPM and expanded support for organic agriculture.^[5,6] The IPM approach has increasingly, in some form, been included in national government policies in the developing countries. On a small-scale, nationally based non-governmental organizations (NGOs), the international organic movement,

^bSee www.fao.org/globalipmfacility/home.htm for an update on activities.

and even some governments, for example the government in Costa Rica, promote organic farming.^{[5]c}

Targeting the Worst Types of Pesticides

The strategy to reduce the worst types of pesticides includes various measures that aim to regulate individual pesticides, for example:^d

- Research and collection of data on toxicity and exposure.
- Classification of pesticides according to hazardousness.
- Developing and publishing risk assessments.
- Monitoring of residues of individual pesticides in agricultural products, environmental media, and human body tissues.
- Registration procedures with decisions on which pesticides should be used in the country.
- Identification of specific pesticides for measures in international law.

All these activities share the feature that they address pesticides individually, based on assumptions of their specific characteristics and exposure situations. Risk reduction measures can then involve the reduction of use or changing the mode of use of those particular pesticides.

This “type” strategy is prominent at the global and national levels. Much work is done collecting and evaluating data on the risks with individual pesticides, publishing hazard classifications, chemical safety data sheets, etc., and disseminating these to developing countries.^e Another line of activity is the establishment of global standards for pesticide residues in food products and standards for acceptable exposure levels.^f Specific international conventions single out lists of chemicals for information exchange or phase-out.^g

^cFor example, the World Federation of Organic Agriculture Movements (IFOAM) has over 750 member associations in more than 100 countries.^[7] The reasons for these organizations to promote organic farming and for farmers to adopt them is not only seen as a means to reduce risks from pesticide use, but also as means to improve soil fertility, reducing the economic vulnerability of the farmers, etc.

^dWhat types of pesticides are judged to be the “worst” is of course very much a value judgment, depending on priorities made on the risk for the agricultural workers, the consumers, the local or global environment etc., but it is a judgment usually made based on significant amounts of scientific data, when such are available.

^eSee www.who.int/pcs and www.chem.unep.ch for information on activities.

^fSee, for example, www.codexalimentarius.net for information on activities.

^gThis includes the Stockholm Convention on Persistent Organic Pollutants, see www.pops.int and the Rotterdam Convention on the Prior Informed Consent Procedure for Certain Hazardous Chemicals and Pesticides in International Trade, see www.pic.int.

Furthermore, much of the capacity building from the UN agencies is geared toward giving countries the legislation and human resources to implement registration schemes.^h At national level, the registration systems that are built up in many developing countries give them the capacity to refuse import, ban, or restrict individual pesticides.

Improving the Mode of Using Pesticides

The strategy to improve the mode of using pesticides encompasses promoting measures that aim to ensure reduced exposure. A diligent, or as it is mostly called “safe,” mode of handling and using pesticides requires farmers to adhere to guidelines such as:

- Storing the products away from food and reach of children.
- Following the correct timing and dose of application including the observance of preharvest intervals.
- Having proper spraying equipment that does not leak.
- Using protective garments during mixing and spraying.

At the global level, a number of guidelines on how to transport, handle, dispose, etc. pesticides in the safest way are produced by UN agencies. The most important one is the International Code of Conduct on the Distribution and Use of Pesticides, which is widely adopted by governments and industry.^{[8]i} International NGOs support this Code and monitor compliance to it.^j At the national level, most efforts to ensure that farmers and workers adopt safety measures are educational rather than regulative. This includes making safe use training part of the extension message. Still, national laws are often in place, which forbid farmers to apply pesticides other than in the manner described by the instructions on the label.^k

CHOOSING STRATEGY

The views on which of the different strategies is preferable vary among different groups of stakeholders. In broad terms, the pesticide industry favors the mode

^hSee www.fao.org/ag/agp/agpp/pesticide for information on activities.

ⁱThe leading pesticide industry association Crop Life International has adopted it, as has the national branches of industry associations in many countries.^[5]

^jOne example of such an NGO is the Pesticide Action Network, see www.pan-international.org.

^kThis is, for example, the case in Kenya and Costa Rica.^[5]

Table 1 Comparing the institutions of risk reduction strategies

| Strategy | Institutional category | Examples | Proneness to change |
|----------|------------------------|--|--|
| Use | Constitutional-type | Favored type of agricultural system (including system of pest management) | Very difficult to change, requiring significant political will at global and national level, involving multiple sectors and actors |
| Type | Collective-choice | National, regional, or international laws and regulations banning or restricting certain pesticides | Moderately difficult to change with large variation in time and resources required Relatively easy to implement as it involves few sectors and actors |
| Mode | Operational | Global (or national) guidelines for safe use, such as codes of conduct, label instructions, or safety sheets | Relatively easy to change with few resource and within a short time span Difficult and costly to implement/enforce at local level, particularly at global scale |

Operational institutions provide structures for day-to-day decisions, collective-choice institutions indirectly affect operational choices, and constitutional-type institutions determine rules to be used in designing the collective-choice rules. This categorization of institutions, or rules as they are also called, has been developed for analyzing common pool resource management. (From Refs.^{[11,12].}) (Modified from Ref.^{[13].})

strategy, many NGOs the use strategy, and governments and international organizations the type strategy. The reasons for the different preferences can be attributed to diverse knowledge and value judgments. Knowledge judgments relate to the causal and contributing factors for the risks, the inherent toxic properties of pesticides, and the exposure situation under conditions of use. Value judgments concern what category of risks should primarily be addressed, the acceptable level of risk, and who, at which level of governance, should be responsible for addressing the risks.

At the foundation for discussing the varying judgments lie the two opposing assumptions:

“All pesticides pose risk” vs. “All pesticides are safe, if used as prescribed.”

If all pesticides are considered toxic irrespective of how they are handled, then the use of pesticides per se is seen as the major contributing factor for negative effects. Reducing the use of all chemical pesticides, irrespective of type, would be a consequential risk reduction strategy. If one holds to the other assumption, that all pesticides are safe if they are used as prescribed, the major risk reduction strategy would be geared at making sure they are used according to the safety instructions.¹ This draws attention to the situation in developing countries where a rationale for

promoting use reduction has been the view held by some, and highly disputed by others, that it is unfeasible that any kind of pesticides could be used safely by poor farmers in these areas.

When choosing between risk reduction strategies, in addition to identifying what and who is culpable for the risks, and matching these with normative considerations of who should feel responsible to take action, it is also a question of who has the capacity to act. Which of the strategy(ies) are more effective and efficient? The strategies entail different implementation measures, engage different stakeholder groups, necessitate different institutions in place, and pose different needs for access to knowledge among stakeholders. There are critical voices on the effectiveness of the short-term type of training provided in safe use projects.^{[5,9]m} Meanwhile a more long-term training in IPM or organic farming requires much more human and financial resources. Some see safe use campaigns as an opportunity to encourage the use of pesticides. On the other hand, reducing the use of pesticides without alternative pest management technologies often brings economic losses for the farmers. The continuing switch in the market from older broad agent pesticides to more sophisticated and sometimes less toxic products also increases the price, thus making them less available to the farmers.

Some of the knowledge and value judgments discussed above are embedded in institutional constraints. Institutions influence incentives and support

¹“Safe” in this case could refer to both health and environmental risks, but the concept “safe use” mostly stresses the health risks. In principle, an astronaut dressed sprayer could avoid being exposed to pesticides (even if this does not correspond to reality), but the environment would still be exposed as per definition pesticides are applied in the environment.

^mThere is no global data, and very limited national data, on what proportion of farmers and workers in developing countries receive this training or even less to what degree adopt the precautionary measures. The numbers are, however, likely to be low on both accounts.

information flows. Institutions—here defined as systems of rules or settled practices, formal or informal^[10]—play a vital role in all three risk reduction strategies. However, it can be argued that each strategy is primarily supported by a specific category of institutions as illustrated in Table 1.

The aim of the mode strategy is to establish detailed institutions of safe handling and use, without changing the existing system of pest management. These changes are made by a handful of experts and government officials but it is the multitude of farmers and workers who ultimately determine the degree of implementation.

In the type strategy, there are collective-choice institutions created at the higher governance levels, on which pesticides should be allowed or withdrawn from the market. A number of stakeholder groups are involved, or try to become involved, in making these choices—industry, governments, IGOs, and NGOs. The most enforceable institutions, however, are made by national governments. The implementation of this risk reduction strategy involves a smaller number of people such as customs officers and pesticide retailers.

The use strategy requires changes in constitutional-type institutions; however, it is no formal constitution that makes farmers chose agricultural system—one dependent on pesticides, one less dependent on pesticides, or one completely independent of pesticides. It is institutions like long-term habits, education, training, and attitude, which are decisive and these are in turn created by global structures that favor one type of agriculture system over others.

This categorization of institutions is helpful when comparing the effectiveness and efficiency of the strategies as the categories vary in how prone (and thus costly and/or time consuming) they are to change. The discussion above on the type and number of actors involved in creating or implementing the institutions gives one good basis for evaluation of the resources needed. Likewise, observations in other management areas show that operational institutions usually require the least time to change, collective-choice institutions require more time, and constitutional-type institutions, finally, are the most time consuming to change.^[14]

CONCLUSIONS

Choosing risk reduction strategy and specific measures is a complex matter involving questions about culpability, responsibility, and capacity. Different stakeholder groups come to different conclusions on these issues and thus usually favor either of the use, type, or mode strategy. The three risk reduction strategies involve institutional creation and change at different levels with significant variation in human, financial, and time resources required. In the end, pesticides are

used because they are considered to provide benefits in controlling pest organisms and reduce loss both in quantity and quality of crops and thus increase the economic gains for both the farmer and the country. It is in relation to this that all risk reduction strategies are made.

REFERENCES

1. Scala, R.A. Risk assessment. In *Casarett and Doull's Toxicology, The Basic Science of Poisons*; Amdour, M.O., Doull, J., Klaassen, C.D., Eds.; Pergamon Press: Oxford, 1991.
2. Lacher, T.E.; Goldstein, M.I. Tropical ecotoxicology: status and needs. *Environ. Toxicol. Chem.* **1997**, *16* (1), 100–111.
3. WHO. *Public Health Impact of Pesticides Used in Agriculture*; World Health Organization: Geneva, 1990.
4. Wesseling, C.; McConnell, R.; Partanen, T.; Hogstedt, C. Agricultural pesticide use in developing countries: health effects and research needs. *Int. J. Hlth. Serv.* **1997**, *27* (2), 273–308.
5. Karlsson, S. Multilayered Governance. Pesticides in the South—Environmental Concerns in a Globalised World. Ph.D. dissertation, Department of Water and Environmental Studies, Linköping University: Linköping, Sweden, 2000.
6. FAO. Action taken by FAO on organic agriculture 1997–2003; Food and Agriculture Organization of the United Nations: Rome, 2003; http://www.fao.org/organicag/doc/dg_report_oa.htm. (accessed April 2005).
7. IFOAM. *Organic Agriculture Worldwide. IFOAM Directory of Member Organisations and Associates 2005*; International Federation of Organic Agriculture Movements: Bonn, Germany, 2004.
8. FAO. *International Code of Conduct on the Distribution and Use of Pesticides, Revised Version*; Food and Agricultural Organization of the United Nations: Rome, 2003; www.fao.org/ag/agp/agpp/pesticid. (accessed January 2004).
9. Kimani, V.W. Studies of Exposure to Pesticides in Kibirigwi Irrigation Scheme, Kirinyaga District. Ph.D. dissertation, Department of Crop Science University of Nairobi: Nairobi, 1996.
10. Young, O.R. *Institutional Dimensions of Global Environmental Change Science Plan*; IHDP Report No. 9; International Human Dimensions Programme on Global Environmental Change: Bonn, Germany, 1999.
11. Gibson, C.C.; Ostrom, E.; Ahn, T.K. The concept of scale and the human dimensions of global change: a survey. *Ecol. Econ.* **2000**, *32* (2), 217–239.
12. Ostrom, E. *Governing the Commons, The Evolution of Institutions for Collective Action*; Cambridge University Press: New York, 1990.
13. Karlsson, S.I. Agricultural pesticides in developing countries: a multilevel governance challenge. *Environment* **2004**, *46* (4), 22–41.
14. Ostrom, E.; Burger, J.; Field, C.B.; Norgaard, R.B.; Policansky, D. Revisiting the Commons: local lessons, global challenges. *Science* **1999**, (284), 278–282.

Strawberry Arthropods: Ecology and Control

Noubar J. Bostanian

HRDC, Agriculture and Agri-Food Canada, St. Jean-sur-Richelieu, Quebec, Canada

Joseph Kovach

Department of Entomology, IPM Program, Ohio State University, Wooster, Ohio, U.S.A.

INTRODUCTION

The modern strawberry, *Fragaria x ananassa* Duchesne was created in Europe in the middle of the 18th century by hybridizing the North American *F. virginiana* Duchesne with the South American *F. chiloensis* (L.). Since then, the modern strawberry has become a delicious dessert fruit, with exceptional qualities for processing jams, ice cream, and cake mixes. The cultivated strawberry plant adapts well to different environmental conditions. Field and berry quality are affected by the interaction of environmental factors such as temperature, photoperiod, pests, soil conditions, and fluctuations in air and soil moisture. Although the genus *Fragaria* shows a wide range of regional adaptation, plants of a particular cultivar may develop satisfactorily in one area but less satisfactorily in another.

Major challenges continue to face the strawberry industry. Consumers are demanding high-quality fresh fruit throughout the year. This demand contributed to the development of day neutral strawberry cultivars that produce berries throughout the entire growing season. Concerns for food safety, contamination of the environment, cancellation of certain registered pesticides, and the development of resistance strains among pests have stimulated the development of innovative pest management systems. These systems require greater knowledge by growers and researchers of the biology of the host, pests, vectors, and associated plants.

This article, without claiming completeness, examines the advances made in the management of the principal arthropod pests of strawberries on a worldwide scale. It is our hope that it will generate enough interest in the reader to pursue additional information currently available in the literature.

ARTHROPOD PESTS OF STRAWBERRIES

***Agriotes obscurus* (L.) (Dusky Wireworm)**

In Canada (2000), rows of wheat (*Triticum aestivum* L.) planted eight days in advance of intercropped rows

of strawberries aggregated wireworms at the wheat rows and reduced the mortality of strawberry plants to only 5.3% compared to 43% in the control.^[1]

***Anthonomus rubi* Herbst (Strawberry Blossom Weevil)**

Pheromone monitoring was more reliable than counting clipped buds in the Russian Federation (1996). This pest attained the action threshold of 5% damage buds very quickly.^[2] The level of damage depended on the duration of the budding stage as only the buds served as oviposition sites. Late cultivars, e.g., “Pandora” escaped damage. Cultivars with long inflorescences suffered less damage than those with short ones.^[3] In Britain (2004), a blend of Grandlure I and II and lavandulol with a 1:4:1 ratio was effective in predicting the severing damage caused by *A. rubi* by a week. Trapping-out with 1000 traps was unsuccessful.^[4]

***A. signatus* Say (Strawberry Bud Weevil)**

In Canada (1999), overwintering adults appeared when 300 DDs (degree-days) had accumulated from April 1 at temperatures above 0°C. The maximum abundance occurred between 500 and 600 DDs. The summer generation attained its peak from 1250 to 1650 DDs, and a pesticide treatment at that time reduced clipped buds the following season.^[5] In U.S.A. (1999), primary and secondary flower buds removed by hand (similar to buds clipped by this weevil) did not affect yield. Compensation to bud removal was achieved by increased weight of remaining berries. However, clipping tertiary and higher-order buds decreased yield.^[6]

***Chaetosiphon fragaefolii* (Cockerell) (Strawberry Aphid)**

In Japan (1981), covering “Hokowase” strawberry plants with white cheesecloth during runner production reduced viral infection rate.^[7] In laboratory studies (U.K. 2001), the carabids *Pterostichus melanarius* Illiger and *Calathus fuscipes* (Goeze) found

abundantly in strawberry fields consumed a mean of 9 and 12 aphids.^[8]

***Frankliniella occidentalis* (Pergande) (Western Flower Thrips)**

The use of *Orius laevigatus* (Fieber) and *Neoseiulus cucumeris* (Oudemans) in Italy (1992) was successful as biocontrol agents for this pest. Powdery mildew on strawberries should be controlled with fungicides that are not toxic to these predators.^[9] In southwestern France (1998), a misting technique was developed for strawberry production in plastic tunnels that kept the population density of the western flower thrips and *Tetranychus urticae* Koch below their action threshold.^[10] In Australia (2002), two treatments with spinosad combined with releases of *Typhlodromus montdorensis* (Schicha) maintained low thrips numbers on berries during mid-to-late summer.^[11]

***Lygus hesperus* Knight (Western Tarnished Plant Bug)**

In U.S.A. (2000), *Anaphes iole* Girault provided 64% control of this pest.^[12] Presently, attracting this pest toward rows of alfalfa planted at every 40 rows of strawberries appears to be a promising strategy for managing this pest in California.^[13]

***L. linoelaris* (Palisot de Beauvois) (Tarnished Plant Bug)**

The action threshold for this pest was established at 0.15 nymphs per flower cluster in Canada (1990). A sequential sampling program was also developed for monitoring purposes.^[14]

***Otiorhynchus ovatus* (L.)/*O. sulcatus* (Strawberry Root Weevil/Black Vine Weevil)**

In U.S.A. (1984), wild beach strawberries, *F. chiloensis*, were compared to commercial strawberries, *F. x ananassa*, for tolerance to feeding by these two weevils. Weevils fed less and had lower fecundity on *F. chiloensis* leaves. *F. chiloensis* also increased the preoviposition period of newly emerged adults.^[15]

***O. sulcatus* (F.) (Black Vine Weevil)**

In potted plants in Australian nurseries (1981), the entomopathogenic nematode (*Heterorhabditis heliothidis*) parasitized 87% of the *O. sulcatus* larvae.^[16] In Germany (1984), also in potted strawberry plants, the

entomopathogenic fungus *Metarhizium anisopliae* reduced adults by 93%.^[17] At soil temperatures above 12°C (Germany, 1994), larvae, pupae, and young adults were parasitized by entomopathogenic nematodes *Heterorhabditis* sp. Host density decreased by 81–100%. The fungicides benomyl, fosetyl, iprodion, metalaxyl, prochloraz, propamocarb, and pencycuron had no effect on the nematode.^[18] In Britain (1998), a dried rice formulation with any of the following entomopathogenic fungi, *Beauveria bassiana*, *Paecilomyces farinosus*, and *M. anisopliae*, were effective against 1st instar larvae in potted strawberry plants.^[19] The entomopathogenic nematode *Steinernema kraussei* was significantly more effective than *S. carpocapsae*, commercially available.^[20] In Poland (2004), the entomopathogenic nematode, *H. megidis*, was unreliable. It was effective in a cool and wet season and ineffective in a hot and dry season.^[21]

***Spodoptera litura* F. (Cluster Caterpillar)**

Laboratory and greenhouse studies (Japan, 1987) showed that nuclear polyhydrosis virus was slow but very effective in controlling this pest. It was still effective 19 days after application.^[22] In India (1999), the nuclear polyhydrosis virus with boric acid (0.1%) as a sunlight protectant caused 91.7% larval mortality of this pest.^[23]

***T. urticae* Koch/*Phytonemus pallidus* (Banks) (Two-Spotted Spider Mite/Cyclamen Mite)**

Amblyseius reductus Wainstein achieved over 90% phytophagous mite control in the Russian Federation (1990). It was reared outdoors on forest strawberry *F. elatior*.^[24] Laboratory studies from U.S.A. (1998) reported net predation on *P. pallidus* by *T. pyri* Scheuten, *N. fallacis* (Garman), *N. californicus* (McGregor), *A. andersoni* Chant, and *Galendromus occidentalis* (Nesbitt). In the field, *N. fallacis* gave more rapid control of *P. pallidus* and *T. urticae* than *N. cucumeris*. The latter gave longer-term control at lower densities.^[25] In U.K. (2004), *Phytoseiulus persimilis* Athias-Henriot and *N. californicus* were found more often on older leaves with *T. urticae*. Whereas *N. cucumeris* and *N. aurescens* (Athias-Henriot) were prevalent on unopened leaves and fruiting clusters with *P. pallidus*. Feeding studies showed *N. californicus* preferred *T. urticae* to *P. pallidus*. *P. persimilis* did not consume *P. pallidus*.^[26]

***T. urticae* Koch (Two-Spotted Spider Mite)**

In France (1985), a combination of *P. persimilis* with *Cynodromus chilensis* Dosse = *N. californicus* was

very effective. *P. persimilis* had a shock effect on the prey whereas *C. chilensis* maintained the pest at low densities.^[27] In Canada (1990), *Aphidoletes* sp., *Stethorus punctum picipes* Casey and *N. fallacis*, responded numerically to introductions of *T. urticae*. *N. fallacis* responded to the greatest degree. It was resistant to endosulfan and malathion but susceptible to carbofuran and dimethoate.^[28] In Italy (1990), two releases of *P. persimilis*, one in early December and another in mid-February, kept *T. urticae* populations near zero until June.^[29] In Mexico (1994), cultivars responded differently to *T. urticae* infestations. “Floridabelle” had the lowest density and least damage. “Rainier” had the most damage whereas “Chandler” and “Selva” were intermediate.^[30] In U.S.A. (2002), the greatest decrease in yield in short-day genotypes (“Chandler”) from *T. urticae* feeding was in June. In day neutral genotypes (“Selva”) the greatest decrease was in April and May. This is postulated to be owing to the day-neutral genotype’s more complex flowering responses.^[31]

CONCLUSIONS

All the principal pests of strawberry reviewed here are polyphagous and along with their natural enemies occur on other hosts especially the Rosaceae. Strawberry production methods (open fields, plastic tunnels, mulching, plastic culture, hydroponic techniques, day neutral cultivars, etc.) have pronounced effects on the pests and their enemies. All the pests reviewed have natural enemies, though some such as mirid bugs have few. However, very few natural enemies have been identified as key limiting factors in pest populations in commercial operations. Furthermore, because strawberry is a high-value crop and grown as an annual or a short-term perennial crop, relying on natural populations for pest control is difficult. Consequently, though considerable progress has been achieved at the research level in recent years, only phytophagous mites are managed by different species of phytoseiids worldwide. Other biocontrol methods have been developed, but are too expensive or complicated to implement when compared with the application of conventional pesticides (count and spray). More effort should be channeled to the development of microbial and nematode biocontrol agents. Furthermore, a more comprehensive understanding of the effects of pesticides to non-target beneficials is a must, as it could eliminate the use of pesticides that promote pest resurgences, e.g., phytophagous mites. Finally, habitat management and trap crops should not be forgotten and evaluated whenever possible.

REFERENCES

- Vernon, R.S.; Kabaluk, T.; Behringer, A. Movement of *Agriotes obscurus* (Coleoptera: Elateridae) in strawberry (Rosaceae) planting with wheat (Gramineae) as a trap crop. *Can. Entomol.* **2000**, *132* (2), 231–241.
- Popov, S.Ya. Possibility of monitoring the population density of the strawberry blossom weevil, *Anthonomus rubi* Herbst (Coleoptera, Curculionidae), on strawberry by two methods: counting clipped buds and using pheromones. *Entomol. Rev. (Engl. Transl. Entomol. Obozr.)* **1996**, *75* (9), 104–109.
- Popov, S.Ya. Evaluation of strawberry varieties for resistance to the strawberry blossom weevil *Anthonomus rubi* Herbst (Coleoptera, Curculionidae). *Izvestiya Timiryazevskoi Sel' skokhozyaistvennoi Akademii.* **1996**, *1*, 142–152.
- Cross, J.V.; Hall, D.; Innocenzi, P.J.; Burgess, C.M. Exploiting the sex-aggregation pheromone of strawberry blossom weevil (*Anthonomus rubi*). *Bulletin OILB/SROP* **2004**, *27* (4), 125–132.
- Bostanian, N.J.; Binns, M.; Kovach, J.; Racette, G.; Mailloux, G. Predictive model for strawberry bud weevil (Coleoptera: Curculionidae) adults in strawberry fields. *Environ. Entomol.* **1999**, *28* (3), 398–406.
- English-Loeb, G.; Pritts, M.; Kovach, J.; Rieckenberg, R.; Kelly, M.J. Compensatory ability of strawberries to bud and flower removal: implications for managing the strawberry bud weevil (Coleoptera: Curculionidae). *J. Econ. Entomol.* **1999**, *92* (4), 915–921.
- Kobatake, H.; Sugiura, T.; Minegishi, M. The occurrence of aphid-transmitted strawberry viruses and their control in Nara Prefecture. *Bull. Nara. Agric. Exp. Stn.* **1981**, *12*, 94–108.
- Fitzgerald, J.; Solomon, M. Ground dwelling predatory carabid beetles as biocontrol agents of pests in fruit production in UK. *Bulletin OILB/SROP* **2001**, *24* (5), 155–159.
- Benuzzi, M. La difesa biologica-integrata della fragola. *Informatore-Agrario.* **1992**, *48* (11), 53–57.
- Pommier, J.J. La brumisation sur fraisières. *L'Arboriculture fruitière* **1998**, *513*, 25–28.
- Steiner, M. Progress towards integrated pest management for thrips (Thysanoptera: Thripidae) in strawberries in Australia. *Bulletin OILB/SROP* **2002**, *25* (1), 253–256.
- Udayagiri, S.; Welter, C.S.; Norton, A.P. Biological control of *Lygus hesperis* with inundative releases of *Anaphes iole* in a high cash value crop. *Lygus Summit*, Visalia, California, Nov 28, 2000, Visalia Convention Center: 44–56.
- Swezey, S. Personal communication. University of California: Santa Cruz, California, USA.
- Bostanian, N.J.; Mailloux, G. Threshold levels and sequential sampling plans for tarnished plant bug in strawberries. In *Monitoring and Integrated Management of Arthropod Pests of Small Fruit Crops*; Bostanian, N.J., Wilson, L.T., Dennehy, T.J., Eds.; Intercept Ltd.: Andover, UK, 1990; 81–101.

15. Shanks, C.H., Jr.; Chase, D.L.; Chamberlain, J.D. Resistance of clones of wild strawberry, *Fragaria chiloensis*, to adult *Otiorhynchus sulcatus* and *O. ovatus* (Coleoptera: Curculionidae). *Environ. Entomol.* **1984**, *13* (4), 1042–1045.
16. Bedding, R.A.; Miller, L.A. Use of a nematode, *Heterorhabditis heliothidis*, to control black vine weevil, *Otiorhynchus sulcatus*, in potted plants. *Ann. Appl. Biol.* **1981**, *99* (3), 211–216.
17. Zimmermann, G. Further trials with *Metarhizium anisopliae* (Fungi imperfecti, Moniliales) for control of the black vine weevil, *Otiorhynchus sulcatus* F. on pot plants in the greenhouse. *Nachrbl. Dtsch. Pflanzenschutz.* **1984**, *36* (4), 55–59.
18. Backaus, G.F. Biological control of *Otiorhynchus sulcatus* F. by use of entomopathogenic nematodes of the genus *Heterorhabditis*. *Acta Hortic.* **1994**, *364*, 131–142.
19. Booth, S.R.; Shanks, C.H., Jr. Potential of a dried rice/mycelium formulation of entomopathogenic fungi to suppress subterranean pests in small fruits. *Biocontrol Sci. Technol.* **1998**, *8* (2), 197–206.
20. Willmott, D.M.; Hart, A.J.; Long, S.J.; Edmondson, R.N.; Richardson, P.N. Use of a cold-active entomopathogenic nematode *Steinernema kraussei* to control overwintering larvae of the black vine weevil *Otiorhynchus sulcatus* (Coleoptera: Curculionidae) in outdoor strawberry plants. *Nematology* **2002**, *4* (8), 925–932.
21. Abanowska, B.H.; Olszak, R.; Tkaczuk, C.; Augustyniuk, K.A. Efficacy of chemical and biological control of the strawberry root weevil (*Otiorhynchus ovatus* L.) and the vine weevil (*Otiorhynchus sulcatus* F.) in strawberry plantations in Poland. *Bulletin-OILB/SROP* **2004**, *27* (4), 153–159.
22. Nemoto, H.; Okada, M. Microbial control of the tobacco cutworm, *Spodoptera litura*, on strawberry grown in the greenhouse. *Ext. Bull. ASPAC Food Fer-til. Technol. Center Asian Pac. Reg.* **1987**, *257*, 19–20.
23. Kulkarni, G.G.; Kumar, S.S.R.; Hugar, P.S.; Kulkarni, K.A. Persistence of NPV against *Spodoptera litura* Fab. On strawberry in transitional tract of Karnataka. *Ann. Agri-Bio-Res.* **1999**, *4* (1), 45–47.
24. Malov, N.A.; Tokunova, M.V. Open-air rearing of *Amblyseius*. *Zashch. Rast. Mosc.* **1990**, *6*, 22.
25. Croft, B.A.; Pratt, P.D.; Koskela, G.; Kaufman, D. Predation, reproduction, and impact of phytoseiid mites (Acari: Phytoseiidae) on cyclamen mite (Acari: Tarsonemidae) on strawberry. *J. Econ. Entomol.* **1998**, *91* (16), 1307–1314.
26. Fitzgerald, J.; Pepper, N.; Solomon, M. Interactions among predatory and phytophagous mites on strawberry. *Bulletin OILB/SROP* **2004**, *27* (4), 85.
27. Fournier, D.; Pralavorio, N.; Pourriere, O. Étude du phytoseiide *Cydnodromus chilensis* en vue de son utilisation contre *Tetranychus urticae* en culture protégée de fraisier. *Entomophaga* **1985**, *30* (2), 113–120.
28. Raworth, D.A. Predators associated with the two-spotted spider mite, *Tetranychus urticae*, on strawberry at Abbotsford, B.C., and development of non-chemical mite control. *J. Entomol. Soc. B.C.* **1990**, *87*, 59–67.
29. Battaglia, D.; Borriello, M.; Spicciarelli, R. Biological control of *Tetranychus urticae* Koch by *Phytoseiulus persimilis* Athias-Henriot on protected strawberry in the Metapontum area. *Informatore Fitopatologico* **1990**, *40* (7), 44–46.
30. Gimenez-Ferrer, R.-M.; Erb, W.A.; Bishop, B.L.; Scheerens, J.C. Host-pest relationships between the twospotted spider mite (Acari: Tetranychidae) and strawberry cultivars with differing levels of resistance. *J. Econ. Entomol.* **1994**, *87* (1), 168–175.
31. Walsh, D.B.; Zalom, F.G.; Shaw, D.V.; Larson, K.D. Yield reduction caused by twospotted spider mite feeding in an advanced-cycle strawberry breeding population. *J. Am. Soc. Hortic. Sci.* **2002**, *127* (2), 230–237.

Strawberry Insects and Mites in California: Ecology and Control

Frank G. Zalom

Department of Entomology, University of California-Davis, Davis, California, U.S.A.

Douglas V. Shaw

Kirk D. Larson

Department of Plant Sciences, University of California-Davis, Davis, California, U.S.A.

INTRODUCTION

California produces almost 90% of all strawberries (*Fragaria X ananassa*) grown in the U.S.A. High per acre productivity is possible owing to diverse growing regions, a long production season, intensive management, and high-yielding varieties. However, the same factors challenge growers and their pest managers to optimize productivity and maximize net returns. Because most plantations are transplanted annually and are fumigated, soil insects such as root weevils rarely build in abundance to become significant problems.^[1] Rather, the most damaging arthropods are fruit and foliar feeders, which reduce fruit quality and yield. Insect and mite management begins with the production of high-quality nursery transplants, clean production fields and field borders, monitoring throughout the season, and intervention during the season as is warranted.

MITES

The two-spotted spider mite, *Tetranychus urticae*, is a problem for strawberry growers worldwide. A related species, *T. cinnabarinus*, is an occasional problem in southern California and San Joaquin Valley plantations. Damage by both species is expressed as stippling, scarring, and reddening of the leaves and calyx. At high densities, plants become severely weakened and appear stunted, dry, and red in coloration. Mite feeding is particularly damaging during the first four to five months following Fall transplant. Plants are less sensitive to mite feeding after initial berry set, and can tolerate higher mite densities. Treatment thresholds vary depending on location, time of season, variety, plant vigor, and yield potential. The highest mite densities are often observed after peak fruit harvest, followed by a rapid, natural decline in mite abundance when the plants begin to produce new vegetation.

Strawberry varieties vary in susceptibility to spider mite infestation and tolerance of feeding, although all

varieties are highly susceptible to feeding damage.^[2] Short-day varieties are generally more tolerant than are day-neutral varieties. Plant vigor greatly influences impact of spider mites.^[3] Preharvest chilling determined by nursery harvest date or length of pretransplant supplemental cold storage promotes plant vigor. Plants with inadequate chilling often develop greater mite infestations; however, excessive chilling promotes vegetative growth and may adversely affect the yield. Other factors affecting plant vigor are soil preparation, fumigation, fertilization, and use of polyethylene plastic mulch.

When spider mites reach treatment thresholds, several acaricides are available, which provide excellent control. Carbamate and pyrethroid insecticides, both commonly used in strawberry production, can induce spider mite outbreaks. Application of these disruptive pesticides should be avoided early season to conserve beneficial arthropods.

Predator mites including *Phytoseiulus persimilis*, *Galendromus occidentalis*, and *Amblyseius californicus* are commercially available for growers, and also occur naturally in most production areas. The release of *P. persimilis* into California strawberries is one of the most widespread uses of an augmentative biological control agent among US horticultural crops. It is important to carefully monitor spider mites to determine if they are being maintained below economically injurious levels when predator mites are being released. If spider mites exceed threshold levels, significant yield loss will occur. Other common natural enemies include the minute pirate bug (*Orius tristicolor*), big-eyed bugs (*Geocoris* spp.), and brown lacewings (*Hemoro-bius* spp.).

The cyclamen mite, *Phytonemus pallidus*, is primarily a pest of second-year plantings, but it can be transplanted into first-year fields on infested nursery transplants and by pickers, bees, birds, and equipment. Leaves newly expanding from the crown, which are infested with cyclamen mites, show severe stunting symptoms. Yield is substantially reduced, and infested

plants may die. Transplants can be treated in hot water at 38°C for 30 min, before planting, to kill mites. If any damage symptoms are observed in a production field, the infested plant should be removed from the field and destroyed. Acaricides applied to infested production fields are most effective using a high volume of water sufficient to soak the unfolded leaves and immature flower buds located in the crowns.

LYGUS

The primary insects directly damaging strawberry fruit in California are *Lygus hesperus* and *L. elisus*. Lygus bugs are serious pests of central coast plantations, but are rarely pests in southern California. They feed by puncturing individual achenes, stopping fruit development and resulting in irregularly shaped, cat-faced berries.

Lygus bugs overwinter in weeds along roadways and ditches, in weedy fields, and in other crops, especially legumes. In coastal areas, they begin to lay eggs in January. Nymphs emerge in March and early April. Adults emerging from weeds or other vegetation may migrate into strawberries, when these alternate hosts are removed or become less attractive. Monitoring begins in March on weeds nearby strawberries when Lygus bug nymphs first appear, and in strawberries once adults are observed. A useful way to follow the life cycle of Lygus bugs is with degree-days, which can also be used to predict the treatment timing against the more susceptible nymphal stages.^[1] Treatment thresholds are very low—1 Lygus per 20 plants sampled using a 12-inch beating tray, and 1 Lygus in 10 plants using a suction machine.^[4] Pyrethroid insecticides have been the principal control for Lygus since 1996, but there is concern for the development of pesticide resistance. Naturally occurring predators and parasitoids that attack Lygus nymphs or eggs include big-eyed bugs, damsel bugs (*Nabis* spp.), minute pirate bugs, several species of spiders, and the parasitoid *Anaphes iole*,^[5] but these do not reliably prevent economic damage.

LEPIDOPTERA

The larvae of several Lepidoptera species cause direct fruit damage and may damage young transplants.^[6] The corn earworm (*Helicoverpa zea*) feeds on fresh fruit, but is especially important as a contaminant. Federal tolerance for *H. zea* requires downgrading to juice stock if a single 7 mm or larger larva is found per 17.5 kg of fruit (about 1100 berries). Annual monitoring for *H. zea* is recommended in south coast plantations where first generation larvae attack winter strawberries, but may not be necessary in other areas

of the state with lower pest densities. A *Helicoverpa* pheromone trap baited with a *H. zea* lure is used to monitor moth flight activity. Treatment is recommended when 10 or more are trapped in a week, and eggs are detected on leaves.

The beet armyworm, *Spodoptera exigua*, is common in all California strawberry production areas. Moths often fly into strawberry fields in the Fall to lay eggs on transplants, and emerging larvae can severely damage crowns. Later season, larvae feed directly on the berries. Treatments are most effective when targeting newly emerged beet armyworm larvae, and both microbial insecticides and insect growth regulators are effective against this developmental stage.

Occasional Lepidoptera pests of California strawberries include the black cutworm (*Agrotis ipsilon*), the rough-skinned cutworm (*Athetis mindara*), the garden tortrix (*Ptycholoma peritana*), the cabbage looper (*Trichoplusia ni*), and the saltmarsh caterpillar (*Estigmene acrea*).^[1] Weedy fields tend to attract more moths to lay their eggs, and weed control can significantly impact their densities. Most Lepidoptera can be adequately controlled with *Bacillus thuringiensis* or spinosad sprays when damaging populations are detected early.

WHITEFLIES

The greenhouse whitefly (*Trialeurodes vaporariorum*) has become a major problem for California growers. They are especially problematic where there are overlapping hosts including older strawberries, beans, cucumbers, peppers, tomatoes, and ornamentals, which serve as sources for whiteflies that enter new plantations. Greenhouse whiteflies tend to build up in fall, reaching peak densities in late fall with nymphs emerging from this generation in March. In warm weather, whiteflies can complete a generation in as little as 18 days. Greenhouse whiteflies can vector strawberry pallidosis associated virus (SPaV) and beet pseudo yellows virus (BPYV), members of the genus Crinivirus.^[7] A combination of SPaV or BPYV as well as any of several non-whitefly-transmitted viruses must infect a plant before symptoms of pallidosis-related decline will occur. In areas where greenhouse whiteflies have become an annual problem, preventative soil treatments with a neonicotinoid insecticide at the time of transplanting is necessary.

OCCASIONAL PESTS: APHIDS, THRIPS, AND VINEGAR FLIES

Green peach aphid (*Myzus persicae*), melon aphid, (*Aphis gossypii*), and strawberry aphid (*Chaetosiphon*

fragaefolii) are the primary species of concern to California strawberry growers. Aphid densities peak during late March in production fields, and undergo a natural population decline during May and June. In high elevation nurseries, populations peak in mid-to late summer.^[1] Aphids rarely reach damaging levels in production fields, but occasionally cause yield losses because of honeydew production resulting in contamination by sooty molds. Aphids transmit several viruses that can cause significant economic losses if a plantation remains in the field for several years. While not a serious problem in annual production plantings, viruses are a major concern for nursery production where preventative measures such as weed control in and around fields and treatment when aphids are detected are routinely practiced. A complex of parasitoids and predators help to limit aphid densities in production fields.

The western flower thrips (*Frankliniella occidentalis*) is considered a problem when it causes fruit russeting around the cap and under the calyx, and when feeding on blossoms causes the stigmas and anthers to turn brown and wither prematurely. Other types of fruit bronzing also occur, which are associated with phytotoxicity from sulfur and from plant physiological responses to heat. In spring, thrips move from weeds, ice plant, and other flowering vegetation when they are mowed, stop flowering, or dry up. Strawberry plantations often have a mixed population of thrips, which includes a low percentage of the onion thrips, *Thrips tabaci*.

Vinegar flies (*Drosophila* spp.) are occasional contaminants of frozen strawberries. Their tiny maggots enter fruit when eggs laid on ripe fruit hatch. Vinegar flies are present in most production fields later season when temperatures are warm; so eliminating conditions that lead to infestations make it possible to manage the flies before they cause damage. External sources of flies such as cull piles in adjacent fields or orchards should be eliminated. Removing overripe, damaged, or cracked fruit from the strawberry plantation itself and shortening harvest intervals limits breeding sites for the flies.

CONCLUSIONS

California strawberry growers must effectively manage insect and mite pests to achieve high quality and yield.

Arthropod management begins with clean and vigorous transplants from nurseries, good sanitation in and around production fields, and preventative applications of insecticides at transplanting for whiteflies where they are problematic. Annual plantings and preplant fumigation eliminate most soil arthropods found in other production areas. Monitoring during the season together with use of cultural and chemical controls that are not disruptive of naturally occurring biological controls will maintain the level of California strawberry productivity among the highest in the world.

REFERENCES

1. Gubler, W.D.; Phillips, P.A.; Pickel, C.; Shaw, D.V.; Welch, N.C.; Zalom, F.G. *Integrated Pest Management for Strawberries*; University of California Division of Agriculture and Natural Resources Publications: Oakland, California, 1994.
2. Walsh, D.B.; Zalom, F.G.; Shaw, D.V.; Larson, K.D. Yield reduction caused by two spotted spider mite feeding in an advanced-cycle strawberry breeding population. *J. Amer. Soc. Hort. Sci.* **2002**, *127* (2), 230–237.
3. Walsh, D.B.; Zalom, F.G.; Welch, N.C.; Pickel, C.; Shaw, D.V. Pretransplant cold storage of strawberries: effects on plant vigor, yield and spider mite (Acari: Tetranychidae) abundance. *J. Econ. Entomol.* **1997**, *90* (3), 818–823.
4. Zalom, F.G.; Pickel, C.; Walsh, D.B.; Welch, N.C. Sampling for *Lygus hesperus* Knight (Hemiptera: Miridae) in strawberries. *J. Econ. Entomol.* **1993**, *86* (4), 1191–1195.
5. Udayagiri, S.; Welter, S.C.; Norton, A.P. Biocontrol of *Lygus hesperus* with inundative releases of *Anaphes iole* in high cash value crop. *Southwest. Entomol.* **2000**, (Suppl. 23), 27–38.
6. Zalom, F.G.; Pickel, C.; Welch, N.C. Recent trends in strawberry arthropod management for coastal areas of the western United States. In *Monitoring and Integrated Management of Arthropod Pests of Small Fruit Crops*; Wilson, L.T., Bostanian, N.J., Dennehy, T.J., Eds.; Intercept Press, LTD.: Winborne, England, 1990; 239–259.
7. Tzanetakis, I.E.; Wintermantel, W.M.; Martin, R.R. First report of beet pseudo yellows virus in strawberry: a second crinivirus able to cause pallidosis disease. *Plant Dis.* **2003**, *87*, 1398.

Submerged Aquatic Weeds: Costs and Benefits of Mechanical and Chemical Control

Rohan D.S. Wells
John S. Clayton

National Institute of Water and Atmospheric Research, Hamilton, New Zealand

INTRODUCTION

Mechanical weed control is the removal or disruption of aquatic weeds by mechanical means. Machines used include various dredges (usually a suction dredge), cutters with reciprocating blades (usually boat mounted) or with conveying equipment to collect the cut weed (a weed harvester), rototillers (an underwater rotary hoe), drag bars (dragging a weighted bar with a sharp leading edge at or above sediment level), and diggers (often with a modified bucket or “clam shell”).

Chemical weed control is the reduction of aquatic weed using herbicides, which are chemicals phytotoxic to target species. Some commonly used aquatic herbicides are endothall, fluridone, and diquat.

Which is the best method of weed control? This is a question often asked, but there is not one method best for all situations. There are many factors to consider: the problem being addressed, the weed species and the area targeted, what outcomes are sought, the aquatic ecosystem (biodiversity and impacts of no action and alternatives), the environmental factors (such as wave fetch, water flow, temperature, water clarity, or nature of lake bed), the ease of access, and the budget available. Even within a single water body or waterway there can be multiple issues to address, requiring an “integrated approach” with different options or combination of options used at different times and locations.

MECHANICAL CONTROL

Weed-Cutting/Weed-Harvesting

A weed harvester cuts the weed and collects it on a platform for shore disposal (Fig. 1). Key benefits of harvesting may include a rapid removal of weed from a sensitive site, and a common public perception that harvesting is environmentally preferable to adding chemicals to water. Negative aspects include the potential for weed-cutting machines to spread invasive weeds to new sites since they produce a lot of cut fragments that are not collected and they are very difficult to

decontaminate before entering a new water body. Harvesters are generally suitable for use in only sheltered water where weeds in amenity areas are commonly mowed to a depth of 2 m and where the risk of further weed spread is not relevant. Some sites are unsuited for cutting because of uneven bottom contours, obstacles or high flows, and low water clarity that can make it difficult to view cutting lines. Weed harvesting removes nutrients from the water body (via plant biomass) but in most cases the quantity of nutrients removed is of little consequence as it is less than the inputs from the catchment, and often much less than the nutrients released from the sediments during periods of anoxia or disturbance. Weed harvesting may have to be repeated several times in a growing season and usually results in capture of a wide range of aquatic organisms (including many small fish) that inhabit or take refuge in the weed. In some cases, repetitive harvesting can result in reduced regrowth rates and when exotic weed beds are cut close to the sediment level, a change to a more desirable species can sometimes occur.^[1]

A harvester can be a large capital outlay and has maintenance, operating costs, and an operator to budget for. Cost-effectiveness depends on the amount of use the machine gets and the capacity of the machine to handle the job. Costs can be competitive. For example, one regional authority in New Zealand (Environment Bay of Plenty, East Coast North Island) operates a small weed-cutting boat in drains (>2 m wide and 0.5 m deep) at a cost of \$150/ha (Fig. 2). (Note: Costs are those for New Zealand but quoted in U.S. dollars) This cost includes operating, maintenance, and capital depreciation but not weed removal. A weed-harvester (Fig. 1) cutting, collecting, and dumping usually costs from around \$1000/ha, but this depends on the rate of removal possible and proximity to the unloading point.

Rototilling

Rototilling (under water rotary hoeing) can be used in water depths of between 1.5 and 4 m. The depth of sediment penetration affects the cost and the outcome.



Fig. 1 A weed harvester cuts and collects the weed for shore disposal.

Deep rototilling (3–5 cm sediment depth) is more costly (about \$2500/ha) but provides a greater duration of control (1–2 yr), while shallow rototilling (at sediment surface) is more rapid and cheaper (about \$500–1000/ha) but provides only about one season's control.^[2] Soft sediment texture and absence of obstacles facilitate ease of operation and success of the outcome. Rototilling has more ecological impact than weed harvesting because it removes more plant material and disturbs the sediment.

Draglining/Dredging

Draglining involves towing a heavy chain between two tractors on either side of a drain. This method is less

expensive and effective but requires access to both sides of a waterway. A drag bar is cheaper to operate in narrow channels, as only one tractor is required. Costs vary from \$250 to \$500/ha depending on whether a wheeled vehicle or a tracked vehicle is required and the reach required from the edge of the waterway.

A mechanical digger (with a wide draining bucket) is usually used in waterways where sediment and weed need to be removed. This method often removes large amounts of benthic fauna and fish (particularly eels) associated with the weed and produces high turbidity and sometimes anoxia. Diggers can overdeepen and overwiden drains and often affect the sides of drains causing bank instability and loss of marginal habitat. Also, these machines often spread invasive weeds to new sites. Costs are about \$2500/ha. A mechanical

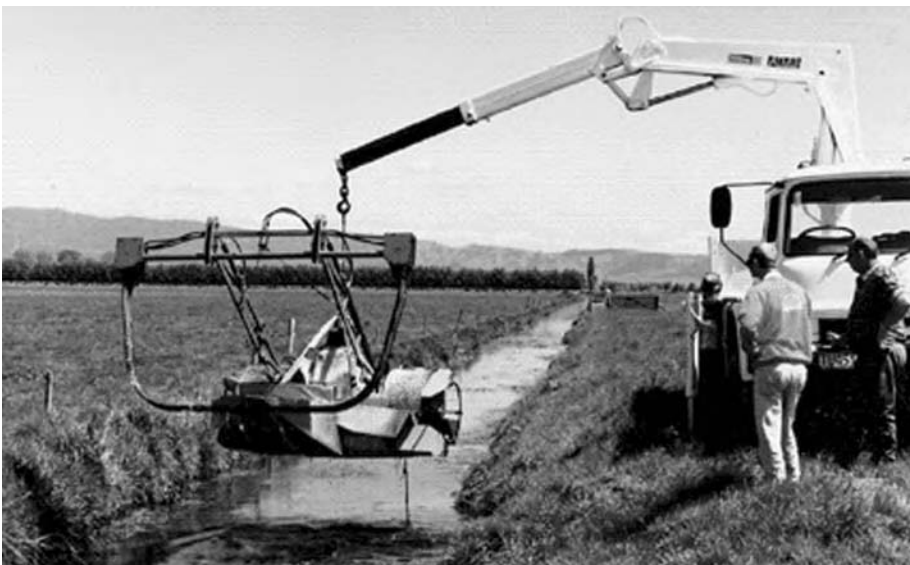


Fig. 2 This weed cutter cuts the weed, which is left to float downstream to the sea. A digger at a screened pump station or at a culvert can remove any cut weed if desired.

digger with a long clam shell is more efficient and has less environmental impact as its clearance per lift can be two to three times greater and it can pull weeds out with minimal disturbance of the bottom sediments. Costs start from about \$1000/ha.

Suction dredging involves sucking up weeds (including roots) into a receptacle, such as a floating barge or fine mesh bags, for later shore disposal (Fig. 3). Few fragments are lost with this method and costs are about \$7–10,000/ha with a clearance rate of up to 20 days/ha in a dense weed. Weed can be eradicated from some sites but reestablishment can be rapid if sites nearby act as a source of reinfestation. The high cost and slow removal rate makes this option unsuitable for general weed control but useful for removal of targeted infestations from areas at an early stage of establishment.

CHEMICAL CONTROL

Endothall, fluridone, and diquat are commonly used aquatic herbicides. In New Zealand diquat is the most commonly used one, as it is very effective on the key target oxygen weeds such as *Elodea canadensis*, *Lagarosiphon major*, *Egeria densa*, and *Ceratophyllum demersum*.^[3] It also does not harm non-nuisance native plant species, such as charophytes, and our native potamogetons and milfoils are little affected and recover rapidly (Fig. 4). For *Hydrilla verticillata*, the potassium salt of endothall is very effective, whereas diquat has no effect. On the other hand, endothall has no effect on *Egeria*, but works on the other target species even in very turbid water, where diquat is adsorbed by negatively charged particles and rendered ineffective. Fluridone has been extensively trailed in New Zealand

in laboratory, mesocosm, and field trials but has no proven benefits here.^[4,5]

Chemical costs are usually low: \$150–\$350/ha depending on susceptibility of the species, and around \$700/ha inclusive of application costs. In most situations, there can also be additional costs for permission to use a herbicide (from statutory authorities) and there may be monitoring requirements. Restrictions may also apply if treated water is to be used for bathing, fishing, drinking, stock-watering, or irrigation and alternative water sources may need to be provided for a short time.

Treated plants remain in the water body and decay in situ. In water bodies of high weed biomass to water volume this may require a maximum of 25% of the water body to be controlled at a time to avoid significant oxygen depletion and nutrient release. Herbicide drift off-target must also be considered and is dependent on the amount of water movement and persistence of the herbicide.

EXAMPLES OF SUCCESSFUL CONTROL

A range of New Zealand examples is given to highlight that no method is “best” for all situations. The following are examples of the best solutions for the particular outcome sought at the time.

Booms and Screen Cleaners on Hydroelectric Lakes

Ceratophyllum demersum (hornwort or coontail—an alien invasive plant in New Zealand) clogs intake screens at the Lake Whakamaru power station. Lake



Fig. 3 The leading craft provides power to a venturi suction pump the intake of which is diver directed. The material is discharged into a large, fine net in the amphibious barge behind.

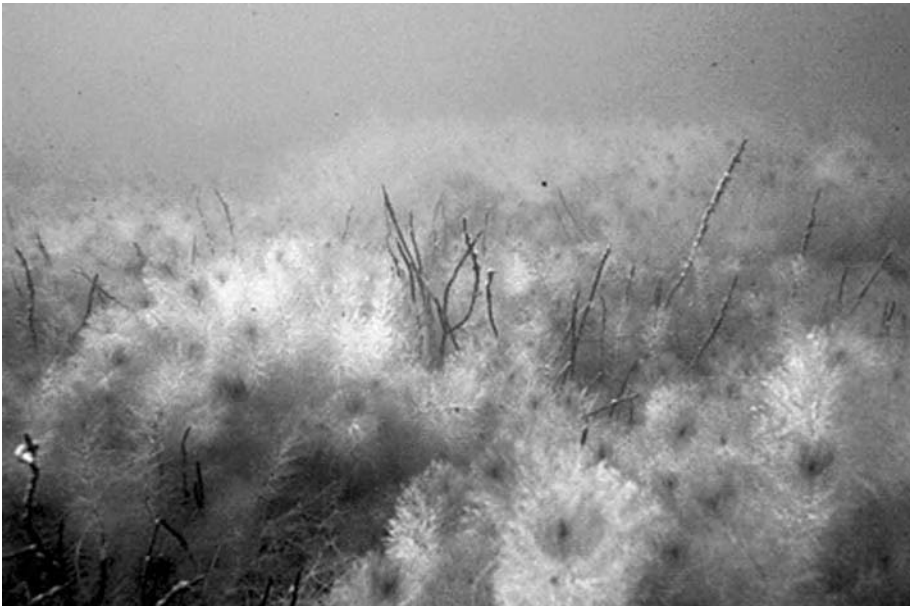


Fig. 4 The “charred” stalks of what was a tall dense lagarosiphon weed bed treated with diquat are evident among native milfoil that has regenerated following the treatment.

Whakamaru (712 ha, on the Waikato River, central North Island) has 237 ha of weed beds and is just one of several hydroelectric lakes that have seasonal problems from submerged weed drift. Weed efflux has a marked seasonality, peaking from April to July (autumn to early winter) with $>10,000/\text{m}^3/\text{yr}$ being removed in some years from the boom and intake screens of Lake Whakamaru alone. Yet this still amounted to $<1\%$ of what was estimated to be growing in the lake. The best solution to date has been to deal with the problem mechanically at the dam with high-capacity automatic screen cleaners and a deflection boom for floating rafts of weed (Fig. 5).

Chemical Control in Flowing Water

The Rangitaiki intake canal (4.2 km long, 20 m wide, and up to 3 m deep) for the Wheao Power Station (located in central North Island, New Zealand) was designed to carry a water flow of $22 \text{ m}^3/\text{sec}$ (cumecs). *E. canadensis* (elodea) established in the canal and grew up to 3 m long and enhanced sedimentation (over 1 m thick) in the top central portion of the canal and reduced maximum flows to 12 cumecs. Initially, the weed and the sediment were dragged from the canal with a blade. The canal is also a prized trout fishery but trout numbers declined markedly with this disturbance regime. Chemical trials established that 20 L diquat (Reglone[®]) applied over two 100 m sections of the upper canal (with flows reduced to 2 cumecs) removed most of the elodea and then higher flows subsequently scoured sediment from the central portion of the canal, restoring its 22 cumec carrying capacity. Chemical application is anticipated once every 3 or 4 yr.

Weed Harvesting in Small Rivers and Drains

Two U.S.-manufactured harvesters have been used on a regular basis to cut and remove nuisance weed growths twice a year from the slow-flowing Avon river, which flows through the city of Christchurch (central east coast, South Island). Local residents have been opposed to any form of chemical control of the dense, often surface-reaching beds of *Potamogeton ochreatus* and *Potamogeton crispus* (pondweeds) that interfere with kayaking, rowing, and aesthetics. In this case, the added cost of mechanical control and weed removal using imported harvesters vs. the use of a chemical was acceptable. Elsewhere, a variety of locally designed cutting machines are operational around the country, mostly purpose-built for use in small water bodies, canals, and drainage systems. Locally built cutters tend to rely on cutting without removal, particularly where cut weed can be removed by flowing water (Fig. 2).

Chemical Control in a Small Recreational Lake

Lake Wiritoa (26 ha sand dune lake located on the southwest coast, North Island) had a diverse native plant flora but *C. demersum* (hornwort) invaded the lake and grew densely up to 4 m tall (often surface-reaching) causing problems for swimming and boating (mainly water skiing). Six hectares of hornwort were sprayed with diquat (at 30 L Reglone/ha) injected at about 1–1.5 m subsurface using trailing hoses. Hornwort was reduced to about 5% of its original abundance within 6 weeks but total vegetation cover was little affected as the native low-growing non-nuisance plants *Nitella* sp. and *Chara australis* (Charophyte



Fig. 5 A weed boom holding back 6 ha of floating hornwort. This weed efflux is a seasonal occurrence.

macroalgae) increased in abundance markedly (with less competition from hornwort). This was a highly desirable result with a major reduction in nuisance weed and a large increase in native plant abundance (Fig. 6). Chemical application may be required annually or perhaps less frequently to maintain a desirable native plant flora and prevent displacement by hornwort.

Suction Dredging in a Large Glacial Lake

Lake Wanaka (180 km², Central Otago, South Island) is a deep (311 m), clear lake dominated by native plant communities to 50 m depth. *L. major*, a tall growing invasive oxygen weed (known in the aquarium plant trade erroneously as “*Elodea crista*”) was first recorded in the lake in 1972 and rapidly established localized tall dense surface-reaching growths displacing native species and affecting recreational activities, as it has elsewhere in a wide range of New Zealand’s lakes. Initially, high-cover areas were sprayed with diquat and then hand weeded using SCUBA divers. Since 1980 suction dredging has been the primary method of weed control, where the plant is removed with its roots to a floating container for onshore disposal (Fig. 3). Outliers and public amenity areas have been the primary target where the objective has been to help reduce further spread within the lake and to minimize the risk of transfer to nearby uninfested water bodies. Regular surveillance of non-infested areas enables early identification of new infestations. This strategy has achieved eradication of several outliers and maintenance of minimal weed biomass in amenity and high-risk areas; however, inconsistent funding, and support from various management

agencies has contributed to escalating spread within the lake.

Long-Term Chemical Control in Several New Zealand Lakes

Diquat has been the primary method of weed control for New Zealand’s longest established submerged

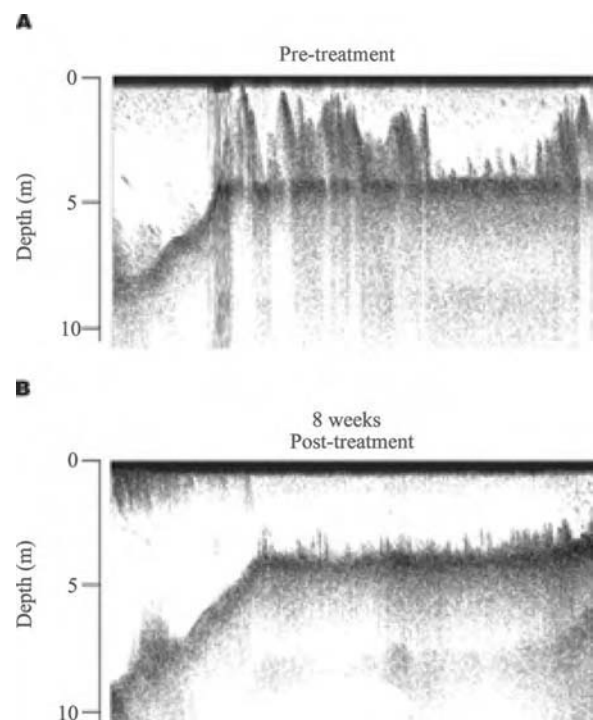


Fig. 6 Sonar traces before and after diquat treatment showing tall surface-reaching beds of hornwort pretreatment and a low-growing native vegetation posttreatment.

aquatic weed problems, such as those experienced in the Rotorua Lakes District (central North Island) since the late 1950s. Extensive areas of nuisance weed can be controlled quickly with chemical application from a boat. This method has been the most cost-effective option for controlling oxygen weed beds on such a large scale.^[6] Selective herbicide treatment with gel-formulated diquat has been effective in controlling target weed species (*E. densa*, *L. major*, and *C. demersum*), and in some instances has enhanced the maintenance of a desirable charophyte vegetation (*Chara* and *Nitella* species), which is resistant to the effects of diquat.^[7] The fate and environmental impact of diquat when applied to aquatic weed beds has been studied both in New Zealand and overseas. The overall nature of these results has been sufficiently favorable to enable continued use for aquatic weed control.

CONCLUSIONS

Even within a single water body or waterway with an aquatic weed nuisance there can be multiple issues to address, requiring an integrated approach with different options or combinations of options used at different times. Simply no one method of aquatic weed control is best for all situations. Each method has its advantages and disadvantages and they need to be weighed up carefully before formulating an aquatic weed management strategy. More information on weed control options can be found on the NIWA website.^[2]

ACKNOWLEDGMENTS

Staff from the Aquatic Plants Group provided valuable comments on this article. The Foundation of Research Science and Technology funded the research program (C01x0221—Aquatic Plant Management) supporting this contribution.

REFERENCES

1. Howard-Williams, C. Processes of aquatic weed invasions: the New Zealand example. *J. Aquat. Plant Manage.* **1993**, *31*, 17–23.
2. NIWA website, <http://www.niwa.co.nz/rc/prog/aquaticplants/weedman/>
3. Clayton, J.S. Review of diquat use in New Zealand for submerged weed control. Proceedings of EWRS/AAB, 7th Symposium on Aquatic Weeds, Loughborough, 1986; 73–79.
4. Wells, R.D.S.; Coffey, B.T.; Lauren, D.R. Evaluation of fluridone for weed control in New Zealand. *J. Aquat. Plant Manage.* **1986**, *24*, 39–42.
5. Hofstra, D.E.; Clayton, J.S. Control of dioecious New Zealand hydrilla using fluridone in mesocosms. *J. Aquat. Plant Manage.* **2001**, *39*, 125–128.
6. Clayton, J.S. Aquatic weeds and their control in New Zealand lakes. *Lake Reservoir Manage.* **1996**, *12* (4), 477–486.
7. Clayton, J.S.; Tanner, C.C. Selective control of submerged aquatic plants to enhance recreational use of water bodies. *Int. Ver. Theor. Angew. Limnol. Verh.* **1988**, *23*, 1518–1521.

Sugarcane Diseases: Ecology and Control

Stuart Rutherford

*Crop Biology Resource Centre, SA Sugarcane Research Institute,
Mount Edgecombe, Kwa-Zulu Natal, South Africa*

INTRODUCTION

Host plant resistance is the most important method for control of sugarcane diseases. Cane breeding, as we know it, began only 100 years ago in response to disease. *Saccharum officinarum* plantations in Java were ravaged by the putative viral disease “sereh” and mosaic.^[1] As no resistant clones of *S. officinarum* were available, interspecific hybrids were developed with a resistant clone of wild *Saccharum spontaneum*. Following backcrossing to *S. officinarum*, the first sereh and mosaic resistant “supercane,” POJ2878, was produced in 1921.

Modern commercial varieties are descended from hybrids of this type and have a narrow genetic base. Pedigrees trace to as few as two *S. spontaneum* clones and a limited number of *S. officinarum* clones used as female parents.^[2] This narrowness has become very apparent, and improvement in certain traits is increasingly problematic. Several diseases pose serious threats to continued widespread sugarcane cultivation. For example, although eyespot is currently of minor importance, new *Bipolaris sacchari* races could conceivably cause an epiphytotic similar to that of southern corn leaf blight by exploiting limited cytoplasmic genetic diversity in sugarcane.^[3] However, this risk may be small, as most varieties appear to have some level of partial resistance.^[4]

Sugarcane varieties begin as single seedlings. Propagation from the initial plant is clonal through the use of stalk cuttings called setts or seedcane. The buds along the stalks germinate to produce new plants. Clonal propagation creates the potential for disease problems, because systemic diseases, within the stalk, can be spread to new fields and are multiplied during the propagation process. Clonal increase can also continue until a variety occupies a large area as a monoculture, again posing increased disease risk from introduced or genetically adapted pathogens.

SEEDCANE QUALITY

Most important sugarcane diseases are systemic. Planting infected seedcane can spread these diseases and they can increase in ratoons—subsequent crops that emerge from the stubble of previous ones. Depending

upon variety, environmental conditions, and management, ratooning may be repeated many times. Eventually, replanting becomes necessary, owing to disease buildup and yield decline. Following plough out, diseases can persist in volunteer regrowth to contaminate newly planted fields.

The planting of healthy seedcane into volunteer-free fields is essential for general disease control. It is important to establish “nurseries” with heat-treated or tissue culture-derived stock to provide healthy seedcane. Hot water treatment, at 50°C for two hours, is essential for the control of ratoon stunting and eliminates some other systemic pathogens including those causing smut and leaf scald, although control of the latter is less complete.

Some 80 diseases are listed on the International Society for Plant Pathology website.^[5] A few of these are considered below.

VIRAL AND PHYTOPLASMAL DISEASES

Fiji Leaf Gall (*Fiji disease virus*, FDV; Reovirus) was first observed in Fiji and is now found in Australia, Indonesia, other Pacific islands, and South East Asia where it is capable of causing a 100% crop loss in susceptible varieties. Symptoms appear as raised off-white galls on the lower surface of the leaf accompanied by severe stunting of the plant. *Fiji disease virus* is transmitted in infected seedcane and by delphacid planthoppers of the genus *Perkinsiella*. Putative vectors are present in many other sugarcane-growing countries. Consequently for FDV-free countries, it is a major quarantine concern when importing sugarcane germplasm from affected countries. Control is accomplished by planting resistant varieties.

Mosaic (*Sugarcane mosaic virus*, SCMV; *Sorghum mosaic virus*; Potyvirus) is found in nearly all sugarcane-growing countries. On leaves, scattered areas of lighter green coloration are most visible on young leaves, particularly near their base. As well as spreading the virus in infected seedcane, aphids also transmit the virus in a nonpersistent manner particularly when migrating populations peak in spring and summer and the cane is young and susceptible to attack. In South Africa, new plantings are at greater risk if established during midspring to midsummer.^[6]

Sugarcane mosaic virus has many graminaceous alternative hosts including sorghum and maize. Host plant resistance is the chief means of control.

Yellow Leaf Syndrome (YLS, *Sugarcane yellow leaf virus*, Luteoviridae; Sugarcane leaf yellows phytoplasma) was first reported in Hawaii in 1989,^[7] and subsequently in Brazil where it has caused extensive yield losses.^[8] To date, it has been reported in more than 30 countries. Yellow Leaf Syndrome has been linked to two systemic phloem-inhabiting pathogens, a phytoplasma that is leafhopper transmitted,^[9] and a luteovirid that is transmitted by the aphids *Melanaphis sacchari* and *Rhopalosiphum maidis*.^[10]

Yellowing of the leaf midrib often occurs while the lamina is still green (Fig. 1). Symptoms also include shortening of terminal internodes and sucrose accumulation in leaf midribs. Similar symptoms can be expressed in the absence of either pathogen, and to confound matters further, infected material is often asymptomatic. Expression is more pronounced during drier and cooler months in mature cane, but there is no single factor that can be correlated with expression in all instances.^[10] The development of a tissue blot immunoassay for detection of the virus has made it possible to screen large numbers of plants accurately and quickly for infection.^[11] However, control measures are lacking.



Fig. 1 Characteristic midrib-yellowing symptom of YLS. (Courtesy of R. A. Bailey, SASRI.)

Diseases caused by other phytoplasmas are largely limited to Asia and include grassy shoot, green grassy shoot, Ramu stunt, and white leaf. Transmission is through infected seedcane and by leafhoppers. Mixed results have been reported on the effectiveness of thermotherapeutic seedcane treatments for eliminating phytoplasmas.

BACTERIAL DISEASES

Xanthomonas albilineans causes leaf scald, which can manifest itself by the sudden wilting and death of plants without the appearance of prior symptoms. In other cases, white streaks appear on the leaves that may coalesce and turn brown. Disease symptoms are associated with the production by the pathogen of a toxin, albicidin. The pathogen can also exist in a latent form, behaving as an endophyte. It is transmitted through seedcane and harvesting equipment, and by wind and rain. Leaf scald is controlled primarily with resistant varieties.

Herbaspirillum rubrisubalbicans, on the other hand, is a mild pathogen causing mottled stripe. It is considered to be a nitrogen-fixing endophyte capable of colonizing all of the tissues of the plant.^[12] There is no report of yield loss; conversely, some benefit owing to nitrogen fixation is suspected.^[13]

Leifsonia xyli subsp. *xyli* occupies the xylem vessels causing ratoon stunting disease (RSD). Transmission is through infected seedcane and from plant to plant on harvesting equipment. A combination of drought and RSD can greatly increase yield loss in intolerant varieties. Ratoon stunting disease readily builds up in ratoons and can remain undetected owing to the absence of obvious external symptoms. Because of this, breeding for resistance has been limited. Disease levels have been reduced through the adoption of management practices, such as fallowing to limit transmission from infected volunteers and crop residues, using hot water-treated or tissue-cultured RSD-free seedcane, and frequently disinfecting harvesting equipment.

A stalk tissue blot immunoassay has been developed, which allows some measure of resistance in terms of percent colonized vascular bundles (%CVB).^[14] Recently, a correlation has been found between yield loss and %CVB (Fig. 2).^[15] This method will allow for more rapid screening for resistance in plant breeding.

FUNGAL DISEASES

Soil-borne fungal pathogens infecting setts when planted have an immediate effect. Pineapple sett rot (*Ceratocystis paradoxa*) infects at the cut ends shortly after planting. The sett becomes hollowed and

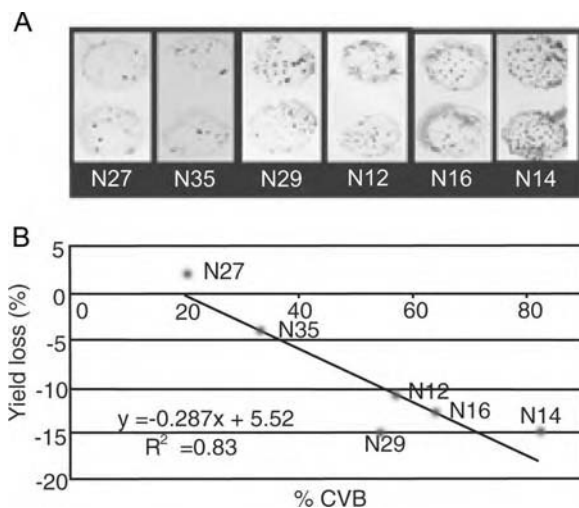


Fig. 2 (A) Tissue blot immunoassay showing vascular bundles colonized by the RSD bacterium for six South African varieties. (B) Relationship between %CVB and yield loss (data points represent six replicates). (Courtesy of S. A. McFarlane, SASRI.)

blackened and may smell like ripe pineapple. Buds do not germinate or shoots may die after emergence. Conditions that delay germination, such as drought or low temperatures, favor disease development. For short sets or after hot water treatment, which increase susceptibility, fungicide treatment can be beneficial.

Fusarium sett rot (*Fusarium verticillioides*) can be differentiated from pineapple sett rot by a more intense red color and absence of pineapple odor. This fungus can also cause rot of standing stalks particularly in association with insect borer damage and may increase the palatability of the cane for the borer *Eldana saccharina* in South Africa.^[16]

Pokkah Boeng is also a *Fusarium* disease (*F. verticillioides* and *Fusarium subglutinans*). Symptoms include chlorosis near the base of the leaf, crumpled, stunted, and twisted leaves, and possible malformation of the stem. In advanced stages, top rot can kill the growing point (Fig. 3). Although outbreaks can be spectacular, plants often recover from symptoms and crop damage is usually not significant.

Smut (*Ustilago scitaminea*) incidence increases following periods of hot dry weather. Spore survival in the soil decreases with moisture. In irrigated areas it is beneficial to irrigate a few weeks before planting to decrease the viable spore load in the soil. New varieties are screened for smut resistance by dipping setts in spore suspensions and eliminating those that subsequently produce whip-like sori (Fig. 4). Whips can release millions of air-borne spores daily, which can both accumulate in the soil and infect standing stalks through the nodal buds. Seedcane should be treated in hot water for 30 min at 52°C with a suitable



Fig. 3 Twisted leaves and stalk affected by Pokkah Boeng. (Courtesy of A. Walton, SASRI.)

fungicide to eliminate systemic infection and to provide some protection to germinating buds.

Outbreaks of rust (*Puccinia melanocephala*, brown rust and *Puccinia kuehnii*, orange rust) are favored by cool and damp conditions. The earliest symptoms are small, elongated yellowish spots that are visible on both leaf surfaces. The spots increase in length, turn reddish brown to brown (*P. melanocephala*), and develop a slight chlorotic halo. Pustules erupt mainly on the underside of the leaves, and in the case of *P. kuehnii*, spores are orange to orange-brown. *P. melanocephala* occurs mainly in young cane whilst *P. kuehnii* is more prevalent in mature cane.

Although fungicides including the new strobilurins are effective, economics dictate that the best means of rust control is to grow resistant varieties. However, brown rust resistance has not been stable in certain varieties, presumably because of rust variants.^[17] The same



Fig. 4 Whip-like sorus produced from the apical meristem of a smut-infected stalk. (Courtesy of K. McFarlane, SASRI.)

is true for orange rust. In Australia, the variety Q124 was resistant to *P. kuehni* and was grown over large areas. In 2000, it suffered heavy infection whilst other partially resistant varieties retained their degree of resistance and continued to develop only limited infections.

CONCLUSIONS

Disease control in sugarcane largely depends on: 1) planting resistant or tolerant varieties; 2) planting a diversity of varieties to reduce the monoculture risk; 3) planting good quality, disease-free seedcane; 4) allowing a fallow, planting a green manure or break crop, and eliminating volunteers before replanting; 5) using fungicides when appropriate; and 6) ploughing out severely contaminated fields.

Additional information on sugarcane diseases and their control can be found in a comprehensive publication produced by the International Society of Sugar Cane Technologists and Centre International de Recherche Agronomique pour le Development.^[4]

REFERENCES

1. Rands, R.D.; Abbott, E.V. Sereh. In *Sugarcane Diseases of the World*; Hughes, C.G., Abbott, E.V.,

- Wismer, C.A., Eds.; Elsevier: Amsterdam, 1964; Vol. 2, 183–189.
2. Tai, P.Y.P.; Miller, J.D. The pedigree of selected canal point (CP) varieties of sugarcane. *Proc. ASSCT*. **1978**, *8*, 34–39.
3. Al-Janabi, S.M.; Honeycutt, R.J.; Peterson, C.; Sobral, B.W.S. Phylogenetic analysis of organellar DNA sequences in the andropogoneae: saccharum. *Theor. Appl. Genet.* **1994**, *88*, 933–944.
4. Comstock, J.C. Eyespot. In *A Guide to Sugarcane Diseases*; Rott, P., Bailey, R.A., Comstock, J.C., Croft, B.J., Saumtally, A.S., Eds.; ISSCT/CIRAD, 2000: 100–103.
5. http://www.isppweb.org/names_sugarcane_common.asp (accessed December 2004).
6. Bailey, R.A.; Fox, P.H. The susceptibility of varieties to mosaic and the effect of planting date on mosaic incidence in South Africa. *Proc. S. Afr. Sug. Technol. Ass.* **1980**, *54*, 1–7.
7. Schenk, S. Yellow leaf syndrome—a new sugarcane disease. *Ann. Rep. Exp. Sta. Hawaiian Sug. Planters Assoc.* **1990**, 38.
8. Vega, J.; Scagliusi, S.M.; Ulian, E.C. Sugarcane yellow leaf disease in Brazil: evidence of association with a luteovirus. *Plant Dis.* **1997**, *81*, 21–26.
9. Cronje, C.P.R.; Bailey, R.A.; McFarlane, K. Update on the occurrence of yellow leaf syndrome in the South African sugar industry and evidence for the causal agent. *Proc. S. Afr. Sug. Technol. Ass.* **1998**, *72*, 81–84.
10. Schenck, S.; Lehrer, A.T. Factors affecting the transmission and spread of sugarcane yellow leaf virus. *Plant Dis.* **2000**, *84*, 1085–1088.
11. Schenk, S.; Hu, J.S.; Lockhart, B.E. Use of a tissue blot immunoassay to determine the distribution of sugarcane yellow leaf virus in Hawaii. *Sugar Cane* **1997**, *4*, 5–8.
12. Olivares, F.L.; Baldini, V.L.D.; Reis, V.M.; Baldini, J.I.; Dobereiner, J. Occurrence of the endophytic diazotrophs *Herbaspirillum spp.* in roots, stems, and leaves, predominantly of gramineae. *Biol. Fertility Soils* **1996**, *21*, 197–200.
13. Nogueira, E.M.; Vinagre, F.; Masuda, H.P.; Vargas, C.; Muniz de Padua, V.L.; da Silva, F.R.; dos Santos, R.V.; Baldini, J.I.; Ferreira, P.C.G.; Hemery, A.S. Expression of sugarcane genes induced by inoculation with *Gluconacetobacter diazotrophicus* and *Herbaspirillum rubrisubalbicans*. *Genet. Mol. Biol.* **2001**, *24*, 199–206.
14. Davis, M.J.; Dean, J.L.; Miller, J.D.; Shine, J.M. A method to screen for resistance to ratoon stunting disease of sugarcane. *Sugar Cane* **1994**, *6*, 9–16.
15. McFarlane, S.A. The relationship between extent of colonisation by *Leifsonia xyli* subsp. *xyli* and yield loss in different sugarcane varieties. *Proc. S. Afr. Sug. Technol. Ass.* **2002**, *76*, 281–284.
16. McFarlane, S.A.; Rutherford, R.S. Beneficial and antagonistic interactions between the stalk borer *Eldana saccharina* and *Fusarium* species from borings in sugarcane. *Proc. S. Afr. Sug. Technol. Ass.* **2005**, *79*, 120–123.
17. Dean, J.L.; Purdy, L.H. Races of sugarcane rust, *Puccinia melanocephala*, found in Florida. *Sugar Cane* **1984**, *1*, 15–16.

Sunflower Diseases: Ecology and Control

Tom Gulya

Northern Crop Science Lab, United States Department of Agriculture,
Agricultural Research Service, Fargo, North Dakota, U.S.A.

INTRODUCTION

Sunflower (*Helianthus annuus* L.) is grown primarily as an oilseed crop, with the greatest production in Russia, Ukraine, the European Union, Argentina, China, and U.S.A. US acreage is primarily in eight midwestern states, with North Dakota being the major producer (Fig. 1).^[1] The achenes are also used for human consumption (referred to as confectionery sunflowers), and in birdseed mixes. Sunflowers are also grown as ornamental flowers. The genetic background of the oilseed, confectionery, and ornamental sunflowers is quite divergent,^[2] which greatly influences their susceptibility to different sunflower diseases. Additionally, control measures practical for a high-value floral crop are much different than those for a field crop.

Sunflower is one of the few crops domesticated in U.S.A. As the entire *Helianthus* genus of 66 annual and perennial taxa is native to North America,^[3] there is a great diversity of sunflower pathogens indigenous to North America on wild *Helianthus*. These wild species also serve as sources of disease-resistance genes.^[4,5] Most sunflower pathogens have been spread globally via seed, but the economic impact of individual diseases varies from country to country. The most serious diseases affecting sunflower worldwide are stalk and head rots caused by *Sclerotinia* species, Phomopsis stalk canker, and Verticillium wilt. In U.S.A., *Sclerotinia* diseases, Rhizopus head rot, Phomopsis stalk rot, and rust are the dominant diseases (Table 1).^[6] Two excellent sources of information on current sunflower research are the semiannual journal *Helia* and the annual *Sunflower Research Workshop* sponsored by the National Sunflower Association, whose proceedings are available at www.sunflowerusa.com/research/default.asp?contentID=70.

Most sunflower diseases are controlled through genetic resistance, with some input from cultural practices. Economics preclude the use of fungicides in all but a few instances, and there is one commercial biocontrol product. Many sunflower pathogens are host specific, which makes rotation a viable disease management tool, but some pathogens such as *Sclerotinia* and *Verticillium* have broad host ranges that render rotation much less effective. This article will cover the major

sunflower diseases; the reader is referred to other reviews for more details.^[6-10] Ref.^[6] contains over 800 references prior to 1997.

SEEDLING DISEASES

Sunflower has relatively few seedling diseases, which may be owing to the practice of seeding late in spring when soils are warm. Diseases affecting seedlings include downy mildew [*Plasmopara halstedii* (Farl.) Berl. and de Toni] and damping-off caused by various *Pythium* and *Phytophthora* species. *P. halstedii* is found worldwide on sunflower, except in Australia, and has been reported on other Compositae genera.^[6,9] The pathogen is unique in that it infects the seedling roots to initiate a systemic, often terminal disease, while airborne spores cause only local lesions. Single, race-specific dominant genes can control downy mildew, but with the occurrence of several dozen races worldwide, this requires knowledge of the predominant races present. Newer molecular methods^[11,12] are being developed to supplement traditional methods to identify resistance genes. Downy mildew is also controlled by the use of fungicide seed treatments, but the fungus has been able to develop resistance to some, such as metalaxyl and mefenoxim. Newer chemistries such as azoxystrobin and fenamidone are being registered in several countries to combat fungicide-resistant strains.^[13] The same fungicides generally have a broad spectrum of activity against other Oomycetes, and thus offer some damping-off control.

FOLIAR DISEASES

While many fungal, bacterial, and viral pathogens cause foliar diseases, the economic impact of foliar diseases is usually minimal except where weather is extremely favorable for disease development. Fungal leaf diseases include rust (*Puccinia helianthi* Schwein.), white rust [*Albugo tragopogonis* Pers. = *Pustula tragopogonis* (Pers.) Thines], and leaf spots caused by several species of *Septoria* and *Alternaria*.^[6] Rust is frequently a yield-limiting factor in North

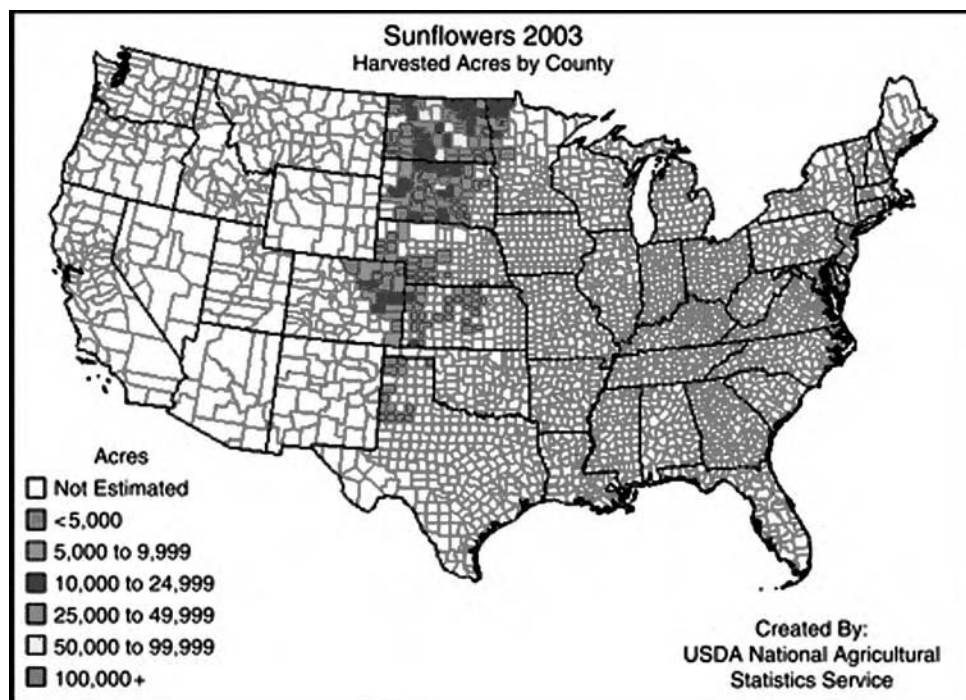


Fig. 1 U.S. sunflower acreage, by county, in 2003. Birdseed sunflower production occurs in all 50 states, but acreages under 5000 are not tabulated by state agricultural statistic services. (From Annual maps available at: <http://www.usda.gov/nass/graphics/county03/sfhar.htm>.)

America and Australia, with confectionery sunflowers being especially susceptible.^[6,10] Both genetic resistance^[14] and fungicides have been used to control rust. White rust, actually an Oomycete and not a true rust, is most severe in South Africa, Argentina, and Australia; it is almost non-existent on sunflower in North

America.^[6] The fungus occurs on a wide range of Compositae weeds, but exists as host-specific races. While the predominant disease symptom is large, raised leaf pustules, the fungus also causes lesions on petioles, stems, and heads.^[15] Control is primarily via genetic resistance. Extensive research on this pathogen has

Table 1 Sunflower disease incidence and severity in the 2003 U.S. sunflower crop, based on a fall survey of 191 fields in eight states

| Disease | Incidence (% of fields) | | Severity (% of crop affected) | Worst states |
|--------------------------|-------------------------|------|-------------------------------|--------------|
| | Range | Mean | | |
| Sclerotinia stalk rot | 2–84 | 2.3 | 22 | ND |
| Sclerotinia head rot | 2–28 | 1.4 | 19 | SD, MN, ND |
| Sclerotinia midstalk rot | 2–40 | 0.7 | 6 | ND |
| All Sclerotinia diseases | 2–84 | 4.4 | 39 | ND |
| Phomopsis stem canker | 2–100 | 2.1 | 22 | CO, KS, MN |
| Rhizopus head rot | 2–28 | 3.9 | 22 | KS, TX |
| Downy mildew | 1–30 | 0.4 | 6 | ND |
| Rust (Puccinia) | 0.1–16 | 1.3 | 44 | SD, TX, CO |
| White rust | 0 | 0 | 0 | — |
| Charcoal rot | 12–26 | 0.3 | 2 | SD, KS |
| Verticillium wilt | 2–40 | 2.1 | 13 | ND, SD |

(From www.sunflowerusa.com/research/research-workshop/documents/Gulya_Disease_Midwest_2003_04.PDF.)

been done by South African scientists. Foliar blights caused by the species of *Septoria* and *Alternaria* are most severe in warm climates with high rainfalls, where defoliation can cause significant yield losses.^[6,16] Otherwise the pathogens are generally confined to lower, senescing leaves. While resistance to these pathogens has been noted,^[17] their relative insignificance worldwide has not spurred plant breeders to incorporate resistance into hybrids. Powdery mildew (*Erysiphe cichoracearum* DC var *latispora* U. Braun) is seen mainly on senescing leaves, and is generally of minimal importance. Bacterial foliar diseases include apical chlorosis (*Pseudomonas syringae* pv. *tagetis* Hellmers) and bacterial blight [*P. syringae* pv. *helianthi* (Kawamura) Young et al.], which are generally of little economic impact.^[6] Sunflower can be infected by over 30 viruses,^[18] but virus diseases are generally only of concern in tropical or subtropical climates, such as in India, where tobacco streak virus is a problem. In North America, viruses are rarely seen on sunflower, with only sunflower mosaic virus noted on wild sunflower in Texas.^[19]

STALK AND ROOT DISEASES AND WILTS

Several broad-host range fungi cause either stalk rot or cankers on sunflower, including *Sclerotinia sclerotiorum* (Lib.) de Bary and *S. minor* Jagger, *Sclerotium rolfsii* Sacc., *Verticillium dahliae* Kleb., and *Macrophoma phaseolina* (Tass.) Goid. *Phomopsis* (*Diaporthe*) *helianthi* (Munt.-Cvet. et al.) is very devastating in Europe; this pathogen is specific to sunflower.^[8] *S. sclerotiorum* and *S. minor* form sclerotia, which overwinter in the soil.^[6,8] The sclerotia germinate myceliogenically to infect the roots of sunflower and other Compositae weeds; no other *Sclerotinia* hosts are prone to root infection. *Sclerotinia* infection progresses up the root system to the basal stalk, where a girdling lesion forms, and the plant wilts and quickly dies. Control of *Sclerotinia* root infection is largely dependent upon genetic resistance, which is polygenic; fungicides are either ineffective or cost-prohibitive. Newer molecular techniques have been developed to help identify *Sclerotinia* resistance.^[20] Cultural control through rotation with non-hosts will reduce sclerotial levels in the soil, but many years are required to be effective. Many *Sclerotinia* mycoparasites have been identified,^[21] and *Coniothyrium minitans* Campbell has been used as a commercial biocontrol agent.^[22] Addition of the mycoparasites to infested fields will hasten sclerotial degradation, but the biocontrol agent is not used as a preventative treatment. *Phomopsis* stem canker originates as a foliar infection that progresses down the petiole to the stem, where a large brown lesion develops while the fungus destroys the pith tissue,

often resulting in lodging, and substantial yield losses.^[8] Resistance is controlled by several dominant genes,^[23] and highly resistant hybrids are available. Fungicides are employed in some European countries, but not in U.S.A. *Verticillium dahliae* is a soilborne fungus that infects sunflower roots, and causes a wilt and leaf mottle that is especially severe on Argentine sunflower. The disease can be controlled by a single, dominant gene, but different strains of *V. dahliae* have been identified, necessitating different resistance genes. The bacterium *Erwinia carotovora* ssp. *carotovora* (Jones) Bergey et al. causes a putrid stalk rot, wilt, often associated with insect damage.

HEAD ROTTS

Fungal and bacterial head rots cause considerable yield losses as they directly impact seed yields, and potentially contaminate seeds with mycotoxins.^[24] Head rots can be caused by *S. sclerotiorum*, *Rhizopus* spp., *Botrytis cinerea* Pers.:Fr., *Phomopsis helianthi*, *Alternaria* spp., and by *E. carotovora*.^[6] Worldwide, head rot caused by *S. sclerotiorum* is the most serious. The fungus produces airborne ascospores that colonize the senescing floral parts during seed filling, but may also infect the back of the receptacle on mature heads.^[8] The fungus may completely disintegrate the head, or it may simply reduce seed number and weight.^[25] A major impact of *Sclerotinia* head rot is the contamination of the harvested seed with sclerotia, which is difficult to separate from the seed and highly undesirable in seed for human consumption. While fungicides can reduce the impact of head rot, application needs to be prior to the onset of symptoms. Resistance to head rot is polygenic, and, unfortunately, is controlled by different genes than those for stalk rot resistance.^[26] Highly resistant commercial hybrids are available, but none give total immunity. The most effective fungicides and the best resistance still allow sclerotia to develop, which makes head rot management in confection sunflowers especially difficult.

CONCLUSIONS

Control of most sunflower diseases is accomplished by genetic resistance, and, in most cases, resistance is conferred by single or several dominant genes. Resistance to *Sclerotinia* stalk rot and head rot is the exception, as resistance is polygenic. Transgenic resistance is being investigated by private seed companies,^[27] but this has not been used in commercial hybrids in any country to date. Traditional methods of identifying sources of disease resistance are being augmented with newer

molecular techniques, and this is especially helpful for polygenic traits such as *Sclerotinia* resistance. Wild *Helianthus* species represent a valuable, and underutilized source of disease-resistance genes.^[4,5] Cultural practices such as plant population, date of planting, and fertilization will have a minimal impact upon disease severity, and are often antithetical with maximizing yield. Tillage of infected plant residue will hasten pathogen degradation, but with the current trend toward minimum or no-till, the use of deep tillage is out of vogue. Biological control has only been commercialized for *S. sclerotiorum*, where its use has been integrated with other control measures. Worldwide, the use of fungicides is largely restricted to seed treatments, whose objective is the control of downy mildew. Foliar fungicides are infrequently employed, and are aimed mainly at controlling rust or *Phomopsis*; disease-forecasting models are being developed to optimize fungicide efficacy. Most sunflower diseases can be adequately managed to minimize yield losses, with the exception of *Sclerotinia* head and stalk rot, which remain the two most challenging pathology research topics. Control of these two diseases will require a concerted, integrated approach, as genetic resistance to this fungus has not resulted in total immunity for any host crop.

REFERENCES

1. National Sunflower Association. 2003 U.S. Sunflower Crop Quality Report; <http://www.sunflowernsa.com/uploads/cqr/cqr2003.pdf>. (Accessed 2004).
2. Cronn, R.; Brothers, M.; Klier, K.; Bretting, P.K.; Wendel, J.F. Allozyme variation in domesticated annual sunflower and its wild relatives. *Theor. Appl. Genet.* **1997**, *95*, 532–545.
3. Heiser, C.B.; Smith, D.N.; Cleverger, S B.; Martin, W.C. The North American sunflower (*Helianthus*). *Mem. Torr. Bot. Club* **1969**, *22*, 1–218.
4. Seiler, G.J. Utilization of wild sunflower species for the improvement of cultivated sunflower. *Field Crops Res.* **1992**, *30*, 195–230.
5. Ronicke, S.; Hahn, V.; Horn, R.; Grone, I.; Brahm, L.; Schanbl, H.; Friedt, W. Interspecific hybrids of sunflower as a source of *Sclerotinia* resistance. *Plant Breed.* **2004**, *123*, 152–157.
6. Gulya, T.; Rashid, K.Y.; Masirevic, S. Sunflower diseases. In *Sunflower Production and Technology*; ASA, CSSA, SSSA: Madison, WI, 1997; 263–379.
7. Gulya, T.J.; Nelson, B.; Lamey, A. Diseases. In *Sunflower Production*; Berglund, D.R., Ed.; NDSU Ext. Ser. Bull. **25**, 1995; 44–63; <http://www.ext.nodak.edu/extpubs/plantsci/rowcrops/eb25w-1.htm> (accessed 2004).
8. Masirevic, S.; Gulya, T.J. *Sclerotinia* and *Phomopsis*—two devastating sunflower pathogens. *Field Crops Res.* **1992**, *30*, 271–300.
9. Sackston, W.E. Downy mildew of sunflower. In *The Downy Mildews*; Spencer, D.M., Ed.; Acad. Press: London, 1981; 545–575.
10. Yang, S.-M.; Dowler, W.M. Sunflower rust. In *Plant Diseases of International Importance*; Chaube, H.S., Kumar, J., Mukhopadhyay, A.N., Singh, U.S., Eds.; Prentice Hall: Englewood Cliffs, NJ, 1992; 345–361.
11. Radwan, O.; Bouzidi, M.F.; Nicolas, P.; Mouzeyar, S. Development of PCR markers for the P15/P18 locus for resistance to *Plasmopara halstedii* in sunflower, *Helianthus annuus* L. from complete CC-NBS-LRR sequences. *Theor. Appl. Genet.* **2004**, *109*, 176–185.
12. Al-Chaarani, G.; Roustae, A.; Gentzbittel, L.; Mokrani, L.; Barrault, G.; Dechamp-Guillaume, G.; Sarraf, A. A QTL analysis of sunflower partial resistance to downy mildew (*Plasmopara halstedii*) and black stem (*Phoma macdonaldii*) by the use of recombinant inbred lines RILs. *Theor. Appl. Genet.* **2002**, *104*, 490–496.
13. Gulya, T.J. Efficacy of single and two-way fungicide seed treatments for the control of metalaxyl-resistant strains of *Plasmopara halstedii* (sunflower downy mildew), The BCPC Conference—Pests & Diseases, 2002. British Crop Protection Council: Brighton, UK, Nov 18–21, 2002; 575–580.
14. Jan, C.C.; Quresh, Z.; Gulya, T.J. Registration of seven rust resistant sunflower germplasms. *Crop Sci.* **2004**, *44* (5), 1887–1888.
15. Kruger, H.; Viljoen, A.; Wyk, P.S. van. Histopathology of *Albugo tragopogonis* on stems and petioles of sunflower. *Can. J. Bot.* **1999**, *77*, 75–178.
16. Kong, G.A.; Simpson, G.B.; Kochman, J.K.; Brown, J.F. Components of quantitative resistance in sunflower to *Alternaria helianthi*. *Ann. Appl. Biol.* **1997**, *130*, 439–451.
17. Carson, M.L. Reactions of sunflower inbred lines to two foliar diseases. *Plant Dis.* **1985**, *69*, 986–988.
18. Brunt, A.A.; Crabtree, K.; Dallwitz, M.J.; Gibbs, A.J.; Watson, L.; Zurcher, E.J., Eds.; *Plant Viruses Online: Descriptions and Lists from the Vide Database*. Version: 20th August 1996. URL; <http://biology.anu.edu.au/Groups/MES/vide/> (accessed 2004).
19. Gulya, T.J.; Shiel, P.J.; Freeman, T.; Jordan, R.L.; Isakeit, T.; Berger, P.H. Host range and characterization of sunflower mosaic potyvirus. *Phytopathology* **2002**, *92*, 694–702.
20. Bert, P.F.; Dechamp-Guillaume, G.; Serre, F.; Jouan, I.; de Labrouhe, D.T.; Nicolas, P.; Vear, F. Comparative genetic analysis of quantitative traits in sunflower (*Helianthus annuus* L.)—3. Characterisation of QTL involved in resistance to *Sclerotinia sclerotiorum* and *Phoma macdonaldi*. *Theor. Appl. Genet.* **2004**, *109*, 865–874.
21. Zhou, T.; Boland, G.J. Biological control strategies for *Sclerotinia* diseases. In *Plant Microbe Interactions and Biological Control*; Boland, G.J., Kuykendall, L.D., Eds.; Marcel Dekker: New York, 1998; 127–156.
22. McLaren, D.L.; Huang, H.C.; Kozub, G.C.; Rimmer, S.R. Biological control of *Sclerotinia* wilt of sunflower

- with *Talaromyces flavus* and *Coniothyrium minitans*. *Plant Dis.* **1994**, *78*, 231–235.
23. Viguie, A.; de Labrouhe, D.T.; Vear, F. Inheritance of several sources of resistance to *Phomopsis* stem canker (*Diaporthe helianthi* Munt.-Cvet.) in sunflower (*Helianthus annuus* L.). *Euphytica* **2000**, *116*, 167–179.
24. Castano, F.; Vear, F.; de Labrouhe, D.T. The genetics of resistance in sunflower capitula to *Sclerotinia sclerotiorum* measured by mycelium infections combined with ascospore tests. *Euphytica* **2001**, *122*, 373–380.
25. Chulze, S.N.; Torres, A.M.; Dalcerro, A.M.; Etcheverry, M.G.; Ramirez, M.L.; Farnochi, M.C. *Alternaria* mycotoxins in sunflower seeds: incidence and distribution of the toxins in oil and meal. *J. Food Prot.* **1995**, *58*, 1133–1135.
26. Gulya, T.J.; Vick, B.; Nelson, B.D. *Sclerotinia* head rot of sunflower in North Dakota: 1986 Incidence, effect on yield and oil components, and sources of resistance. *Plant Dis.* **1989**, *73*, 504–507.
27. Hu, Xu.; Bidney, D.L.; Nasser, Y.; Duvick, J.P.; Crasta, O.; Folkerts, O.; Lu, G. Overexpression of a gene encoding hydrogen peroxide-generating oxalate oxides evokes defense responses in sunflower. *Plant Physiol.* **2003**, *133*, 170–181.

Surveillance

David J. Horn

Department of Entomology, Ohio State University,
Columbus, Ohio, U.S.A.

INTRODUCTION

Surveillance is routine monitoring of vertebrate and invertebrate pests, weeds, and plant pathogens in order to determine their distribution and dispersal. Information from surveillance programs is used to make timely decisions in the operation of national and local quarantines. Surveillance is also used to properly time pesticide applications in integrated pest management. Surveillance is increasingly important as international trade becomes more global, with more people and goods moving about than ever before. The importance of surveillance in the United States is underscored by a 1999 presidential Executive Order on Invasive Alien Species. This order proposes to prevent unwanted pest introductions, to detect and respond to incipient populations of invasive species, and to monitor their spread.

TECHNIQUES

Generally, invading pests exhibit three “phases”: arrival, establishment, and spread.^[1] Knowing a pest’s potential for colonization and establishment is especially useful in designing surveillance plans and assessing risks of establishment for specific target pests. The probability that a species will successfully invade a new region is directly related to its ability to disperse and colonize.^[2] Any effective sampling technique can be used in surveillance, although the most useful techniques are those that detect very low numbers of the pest immediately after its arrival in an area. There is a critical interplay between the biology and behavior of an insect and the efficiency of sampling, especially when using traps. Trap placement and design, prevailing weather and response of the insect to the trap all interrelate. The probability of detecting an isolated pest infestation is directly related to intensity of trapping.^[3]

Attractants

The most efficient surveillance techniques involve pheromones, light, or bait to lure insects onto a sticky trap or into a container where they can be identified. Sex pheromones are perhaps the most efficient

attractants for low-density populations. Incipient outbreaks of gypsy moth (*Lymantria dispar*) are monitored with a nationwide network of pheromone traps.^[4] Bark beetles (Scolytidae) are attracted to traps baited with host plant odors such as pinene. Most attractants generally lure insects from a much shorter distance than do pheromones. Methyl eugenol (“Medlure”) is attractive to the Mediterranean fruit fly (*Ceratitis capitata*) although it is produced neither by the fly nor the host plant. Weather and design impact trap efficiency; nearly always, traps catch many more insects during warmer weather.

Direct Assessment of Pests or Damage

Most state departments of agriculture in the United States oversee routine inspections of nurseries and honey bee colonies. These detect potential plant pests and pathogens, or honey bee diseases and mites. Forest entomologists in the United States, Canada, and many other nations travel regular routes, routinely looking for evidence of defoliation, top kill, and tree mortality due to insects or diseases. Such regular ground-based surveys are especially useful in integrated pest management.^[1]

Remote Sensing and Digital Imaging

Satellite imagery and/or aerial photography are useful for surveillance of wide areas when ground-based surveys are impractical because of inaccessibility. Remote sensing can detect incipient outbreaks of forest pests such as bark beetles or spruce budworm. The phenology of many introduced weeds is such that they reflect green earlier or later in the growing season than does native vegetation and thus are detectable via aerial photography; this is helpful in rangeland weed management.^[4] Infestations of the silverleaf whitefly (*Bemisia argentifolii*) are detectable via satellite imagery.^[5] Digital imaging can speed decision-making in the assessment of potential new pests; a digital photograph of a pest detected in the field can be transmitted within minutes to a taxonomic expert for identification.

OPERATIONS

Many serious pests are imported from overseas. More invasive species have been imported into North America or Australia than have gone from there to other continents, but the issue is worldwide. Most pests are transported via baggage, cargo, mail, or vehicles rather than naturally. Surveillance is operational wherever and whenever people and goods move legally across international borders where it is hoped that dispersal of pests can be limited via inspections and local control. The usefulness of surveillance in quarantine enforcement is demonstrated by the annual interception of over 30,000 infested items at ports of entry into the United States. In the United States, federal surveillance activities are coordinated by the U.S. Department of Agriculture Animal and Plant Health Inspection Service, Division of Plant Protection and Quarantine (USDA-APHIS-PPQ) operating under the Federal Plant Pest Act of 1957. Table 1 lists the current surveillance activities of USDA-APHIS-PPQ. Many other nations have similar agencies, e.g., the Plant Pest Survey Unit of Agriculture Canada. USDA-APHIS-PPQ maintains inspection facilities and administers surveillance programs with cooperation from state departments of agriculture. USDA-APHIS-PPQ also administers the Cooperative Agriculture Pest Survey (CAPS) program, a joint effort by federal and state agencies to detect and monitor crop pests and beneficial insects. Surveillance information is coordinated and made available through the National Agricultural Pest Information System (NAPIS).^[2] Once a pest is established, quarantine services monitor low-density populations in order to retard spread. Public cooperation is an essential component of such programs.

Surveillance can be improved by planning for likely introductions. As an example, the Northeast Exotic

Pest Survey Committee (NEPSC) undertakes risk analyses for potential pests not yet intercepted in the United States, based on ecological and economic criteria. The NEPSC also develops and circulates lists of the most frequently intercepted pests to regulatory agencies.^[4]

Emergency Program: Asian Longhorned Beetle

The Asian longhorned beetle (ALB) *Anaplophora glabripennis* was detected in the mid-1990s in Brooklyn and Amityville, NY, and Chicago, IL. The ALB is a pest in the Orient and could have serious impact on hardwood trees in North America. A chemical attractant for ALB has not been developed, so detection relies on visual surveys. Public cooperation is solicited through widespread dissemination of ALB photos and information. When a single ALB is found, all trees within 1.5 km are surveyed for evidence of ALB infestation. Detection of additional beetles extends the surveillance area another 1.5 km. Infested trees are removed and destroyed. Additionally, the ALB surveillance program identifies sites at risk for ALB infestations. These include wood storage areas, processing areas for cut wood, landfills, utility companies, lumberyards, and palette redistribution centers. Annual visual surveys are conducted in and around such areas. Under this emergency program, North American infestations of ALB remain confined to the areas initially infested (as of December 2002).

Pest Containment Program: Gypsy Moth

The Gypsy moth is of European origin and arrived in Massachusetts in 1869. It is currently established in the northeastern United States, south and west to the Carolinas, and Wisconsin. Larvae can defoliate extensive forested areas, resulting in occasional tree mortality and major cosmetic and nuisance damage. If unchecked it is likely to infest the rest of the United States wherever there are deciduous trees. USDA-APHIS-PPQ has undertaken a “slow the spread” program combining pheromone trapping with intensive surveys at the leading edge of the gypsy moth advance. In the United States about 300,000 pheromone traps are deployed annually. Multiple captures of male gypsy moths in a single location trigger an intensive local search for egg masses, whereupon the surrounding area (100–200 ha) is treated with insecticide after permission is obtained from landowners and public hearings have been held. Eradication is declared after 3 years of no additional captures. The success of this program relies upon efficient detectability and effective control techniques.^[4] The female European gypsy moth does not fly so all long-distance dispersal is assisted by humans. An Asian form of the gypsy moth

Table 1 Surveillance programs of USDA-APHIS-PPQ

| Emergency | Pest Programs |
|-------------------------|---------------------------------|
| Asian longhorned beetle | Asian gypsy moth |
| Citrus canker | Chrysanthemum white rust |
| Karnal bunt | Common barberry/black stem rust |
| Plum pox | European larch canker |
| | Fruit fly |
| | Golden nematode |
| | Grasshopper/Mormon cricket |
| | Gypsy moth |
| | Imported fire ant |
| | Japanese beetle |
| | Noxious weeds |
| | Pine shoot beetle |
| | Potato diseases |
| | Sudden oak death |

has been detected in the northwestern United States and Canada, and surveillance must be more aggressive because its female flies.

Routine Inspection

In addition to emergency and ongoing specific pest programs, USDA-APHIS-PPQ and individual states cooperate in the administration of routine surveillance. As noted, all honey bee colonies are inspected annually for pathogens and mites. Bee colonies infected with American foulbrood or other diseases are destroyed to prevent the spread of the pathogen. Nursery stock is inspected annually and all stock must be certified pest-free before interstate transportation in the United States. This prevents the spread of pests (such as the Japanese beetle) associated with soil and roots. Occasionally, such routine surveillance will uncover a newly arrived pest, as was the case of the European pine shoot beetle (*Tomicus piniperda*) discovered in 1992 during a routine nursery inspection in Ohio.

CONCLUSION

As commerce becomes more global more people and goods are moving more quickly than ever between continents, we can expect that new pests will be introduced annually. Recent examples from the United States include the soybean aphid (*Aphis glycines*) into Wisconsin (2000), the citrus longhorned beetle (*Anaplophora chinensis*) into Washington (2001), and the emerald ash borer (*Agrilus planipennis*) into Michigan (2002). The longhorned beetle arrived in ornamental plants but the invasion route of the others is unclear. Surveillance programs will have to develop more rapid responses to such pests, both in detection and in organization of containment programs. Digital

imaging will become more important in making timely identifications. Improved surveillance techniques including more efficient trap design can be expected, resulting from research into the relationship between pest behavior and trap design. A remaining challenge will be interpreting samples with an absence of pests, which may simply reflect low response of pests to a trap.^[3] The future will see increased surveillance at points of origin, so that more commodities and conveyances will be certified pest-free before leaving the native home of a potential pest. International agreements such as the General Agreement on Tariffs and Trades (GATT) are crucial to the success of surveillance internationally.^[6]

REFERENCES

1. van Emden, H.F. Integrated pest management. Ency. Pest. Mgmt. **2002**. Update 1.
2. <http://www.aphis.usda.gov/oa/invasive/invasive.html> (accessed Dec. 2002).
3. Venette, R.C.; Moon, R.D.; Hutchison, W.D. Strategies and statistics of sampling for rare individuals. *Annu. Rev. Entomol.* **2002**, *47*, 143–174.
4. USDA Detecting and Monitoring Invasive Species. In *Plant Health Conference*; USDA APHIS PPQ, 2000. Internet access: http://www.aphis.usda.gov/ppq/emergency_programs/pestdetection/plthlthconf.pdf (accessed Dec. 2002).
5. Brewster, C.C.; Allen, J.C.; Kopp, D. IPM from space: using satellite imagery to construct regional crop maps for studying crop–insect interaction. *Am. Entomol.* **1999**, *45*, 105–117.
6. Kahn, R.P.; Cave, G.L.; Greifer, J.K.; Imai, E. Quarantines and regulations, pest risk analysis, and international trade. In *Insect Pest Management Techniques for Environmental Protection*; Rechcigl, N.A., Rechcigl, J.E., Eds.; Lewis Publishers: Boca Raton, FL, 2000; 305–336.

Sweetpotato Diseases: Ecology and Control

Christopher A. Clark

*Department of Plant Pathology and Crop Physiology, Louisiana State University,
Baton Rouge, Louisiana, U.S.A.*

INTRODUCTION

Sweetpotato, *Ipomoea batatas* (L.) Lam., is the world's seventh most important food crop, with the greatest production in China and tropical regions of Africa, Asia, and South America. The diseases that affect sweetpotato are strongly influenced by the method of propagation of the crop, site-specific factors such as the presence or absence of pathogens in the soil, and how the sweetpotatoes are handled after they are harvested.^[1] Approaches to control of diseases are often determined by these factors.

Wherever sweetpotato is grown, it is propagated by use of vegetative parts of the plant: storage roots and/or vine cuttings. As a result, those pathogens that systemically colonize the plant, especially viruses, are a major concern worldwide. In temperate zone production areas, sweetpotatoes cannot be grown in the field during the coldest months and therefore are propagated by using storage roots from the previous crop to produce vine cuttings (slips) to plant the succeeding crop. In the tropics, where sweetpotatoes can be grown in the field year round, vine cuttings from fields in production are used to plant new fields.^[2,3] In either case, systemic pathogens can accumulate in planting stock, leading to a phenomenon known as "cultivar decline."^[4]

SYSTEMIC PATHOGENS

Progress in identifying and controlling the viruses that infect sweetpotato has lagged behind that for many other crops. This is because few research programs have addressed the problem^[5] and because sweetpotato is a difficult plant from which to isolate viruses, owing to the high concentration of interfering substances such as latex, polyphenols, and polysaccharides. nevertheless, more than 20 viruses have been identified worldwide.^[6,7] The most destructive disease of sweetpotatoes is known as sweetpotato virus disease. It is caused by the synergistic interaction of the aphid-transmitted potyvirus, *Sweetpotato feathery mottle virus* (SPFMV), and the whitefly-transmitted crinivirus, *Sweetpotato chlorotic stunt virus* (SPCSV).

Generally, neither SPFMV nor SPCSV has a substantial effect on sweetpotato yields when present by themselves, but when they infect plants simultaneously, yield reductions of 80–90% have been observed and the disease has been the major limiting factor to sweetpotato production in sub-Saharan Africa. *Sweetpotato feathery mottle virus* is found wherever sweetpotatoes are grown. *Sweetpotato chlorotic stunt virus* was for many years only known to occur in Africa, but recently, SPCSV has been found in several countries in South America. The other viruses found in sweetpotato are not as widely distributed as SPFMV. Although the impact of viruses in Japan, China, and the U.S.A. is not as great as in Africa, it has been sufficient to justify seed programs that utilize "virus-tested" plants derived from meristem-tip culture to produce propagating material for farmers. There is a paucity of information on how to reduce reinfection of these tissue culture-derived plants by viruses once they are planted in the field. In addition to viruses, the bacterial root and stem rot pathogen, *Erwinia chrysanthemi*, can infect plants systemically and can remain latent in plants or stored roots until environmental conditions that favor soft rot development occur.

ROOT-BORNE PATHOGENS

Several fungal pathogens are able to infect storage roots in the field, but do not survive in soil for very long. Notable examples are: the black rot pathogen, *Ceratocystis fimbriata* and the scurf pathogen, *Monilochaetes infuscans*.^[1] These diseases are far more common in the temperate zone, if infected storage roots are used for "seed." These pathogens can be carried into the plant bed and can grow from the "seed" root up onto the sprouts that arise from it, colonizing the portion of the stem below the soil surface. An integrated program of rotating fields out of sweetpotato production for 2–3 yr, selecting disease-free roots for use as "seed," treating seed roots with a fungicide at the time of bedding, and cutting plants at least 2–3 cm above the soil surface is usually sufficient to eliminate these diseases as economic factors in production.

SOIL-BORNE PATHOGENS

Some sweetpotato pathogens persist in soil for many years and can cause disease whenever sweetpotatoes are planted in those soils. There are four noteworthy examples: root-knot nematode, *Meloidogyne* spp.; reniform nematode, *Rotylenchulus reniformis*; the *Streptomyces* soil rot (or pox) pathogen, *Streptomyces ipomoeae*; and the Fusarium wilt pathogen, *Fusarium oxysporum* f.sp. *batatas*.^[1] Root-knot nematode is widely distributed around the world and is particularly damaging in sandy soils. In addition to causing the galls found on feeder roots of many susceptible plants, it can also cause cracking or formation of bumps on storage roots, with the females and egg masses enveloped within the storage root tissue.

Reniform nematode is not as widely distributed, but within the U.S.A., its range is steadily expanding. This nematode is more difficult to diagnose as females do not develop within the storage roots and it does not induce distinctive symptoms on storage roots, but it has become the predominant nematode in some areas where root knot was once important.

Streptomyces soil rot is a disease that develops when infested soils are dry and the pH is above 5.2. The aggressive feeder root rot can cause dramatic reductions in vine growth and yield of storage roots on susceptible cultivars. In addition, storage roots are often misshapen owing to constrictions caused by infection. Fusarium wilt was once a limiting factor to sweetpotato production in the U.S.A. but is uncommon at present.

Streptomyces soil rot and Fusarium wilt have been greatly reduced in importance by deployment of resistant cultivars and are no longer limiting to production.^[8] Resistance is also available to root-knot nematode, but in the U.S.A., resistant cultivars have not displaced the more popular susceptible cultivars. Resistance has not been found in sweetpotato germplasm to the reniform nematode. Nematode control in sweetpotato therefore still relies on use of preplant-applied chemicals including fumigants such as dichloropropene and non-fumigant materials such as ethoprop.

POSTHARVEST PATHOGENS

Under the proper conditions, sweetpotatoes are routinely stored for 8–10 mo or more^[9] and can be stored even longer. Some of the diseases caused by root-borne and systemic pathogens can be carried into storage in roots infected in the field, or can initiate infection through wounds incurred during harvest, such as Fusarium root and stem canker, black rot, and bacterial root rot. In addition, some of the most significant postharvest disease losses are caused by pathogens that enter through wounds incurred during harvest or when

roots are removed from storage and packed for transport to market. Generally, these develop only in the postharvest environment and include: Rhizopus soft rot, caused by *Rhizopus stolonifer* and *Rhizopus oryzae*; charcoal rot, caused by *Macrophomina phaseolina*; and Java black rot, caused by *Lasiodiplodia theobromae*. These diseases can develop either during long-term storage, or when sweetpotatoes are in the marketing chain. Vine removal several days prior to harvest can help reduce skinning injury during harvest and thereby reduce postharvest disease. The chances of losses in long-term storage are reduced by the practice of curing sweetpotatoes immediately after they are harvested.^[9] This process involves keeping them at 28–30°C and 90–97% relative humidity for 4–7 days, which promotes healing of the wounds. In the U.S.A., they are then stored at about 13–15°C and high humidity for several months, after which they are removed from long-term storage, washed for the first time, and repacked in cardboard cartons. New wounds can occur during the washing/packing process and inoculum from roots that decayed during long-term storage can be transferred to healthy roots. Designing handling systems to minimize wounding, including use of containers that protect roots from wounding can reduce disease. It is generally not considered practical to cure the roots again at this time and therefore fungicides such as dicloronitroaniline have been used to protect the wounds from infection by fungi, especially *Rhizopus* species.

CONCLUSIONS

The strategy for controlling diseases of sweetpotatoes is determined by the nature of the particular disease(s). Diseases that are associated with vegetative propagation of the crop are controlled by use of meristem-tip culture to eliminate systemic pathogens, integrated with practices such as crop rotation, careful selection of disease-free “seed” roots, treatment of roots with protectant fungicides, and cutting of slips above the soil line. Soil-borne diseases are controlled by use of resistant cultivars and/or chemical soil treatments to reduce populations of the pathogens. Postharvest diseases are controlled primarily by curing the sweetpotatoes immediately after harvest and treatment with fungicides during the packing process.

REFERENCES

1. Clark, C.A.; Moyer, J.W. *Compendium of Sweet Potato Diseases*; APS Press, The American Phytopathological Society: St. Paul, Mn, 1988; 74 pp.
2. Ames, T.; Smit, N.E.J.M.; Braun, A.R.; O’Sullivan, J.N.; Skoglund, L.G. *Sweetpotato: Major Pests, Diseases, and*

- Nutritional Disorders*; International Potato Center: Lima, Peru, 1997; 153 pp.
3. Lenné, J.M. *Diseases and Pests of Sweet Potato*; Natural Resources Institute Bulletin no. 46; 1991; 116 pp.
 4. Clark, C.A.; Valverde, R.A.; Fuentes, S.; Salazar, L.F.; Moyer, J.W. Research for improved management of sweetpotato pests and diseases: cultivar decline. Proceedings 1st International Symposium on Sweet Potato Food and Health for the Future, Lima, Peru, July 26–29, 2001; Ames, T., Ed.; Acta Horticulturae, 2002; Vol. 583, 103–112.
 5. Horton, D.E.; Ewell, P.T. Sweet potato pest management: a social science perspective. In *Sweet Potato Pest Management, A Global Perspective*; Jansson, R.K., Raman, K.V., Eds.; Westview Press: Boulder, CO, 1991; 407–427.
 6. Moyer, J.W.; Salazar, L.F. Viruses and viruslike diseases of sweet potato. *Plant Dis.* **1989**, *73*, 451–455.
 7. Salazar, L.F.; Fuentes, S. Current knowledge on major virus diseases of sweet potatoes. In *Kyushu National Agricultural Experiment Station, Proceedings International Workshop on Sweet Potato Cultivar Decline Study*, Sept 8–9, 2000; Nakazawa, Y., Ishiguro, K., Eds.; Miyakonjo, Japan, 2000, 14–19.
 8. Clark, C.A.; Dukes, P.D.; Moyer, J.W. Diseases. In *Fifty Years of Cooperative Sweet Potato Research 1939–1989*; Southern Cooperative Series Bulletin no. 369; Jones, A., Bouwkamp, J.C., Eds.; Louisiana Agricultural Experiment Station, Louisiana State University Agricultural Center: Baton Rouge, Louisiana, 1992; 88–105.
 9. Boyette, M.D.; Estes, E.A.; Rubin, A.R.; Sorenson, K.A. The postharvest handling of sweetpotatoes, with construction guidelines for negative horizontal ventilation curing and storage facilities. *Bull. North Carolina Coop. Extension* **1997**, 42 pp.

Synergy with Microorganisms

Albrecht M. Koppenhöfer

Department of Entomology, Cook College, Rutgers University,
New Brunswick, New Jersey, U.S.A.

INTRODUCTION

Synergism (or potentiation) is defined as the joint (or supplemental) action of two agents resulting in a greater effect than the sum of the activities of the agents acting alone. In pest management, the combination of agents can result in antagonistic, additive, or synergistic effects on speed of kill and mortality of a pest. Additive (also termed complementary) effects occur when the agents act independently of each other, i.e., there is no interaction. Synergistic or antagonistic effects occur when the interaction between agents renders the combination more or less effective in control than in the case of an additive effect (Table 1).

CALCULATION OF INTERACTIONS

Unless the mechanism of the interaction is known, the use of the term synergism is based on statistical tests that determine whether the observed effect is significantly higher than the effect expected for an additive effect. The expected additive mortality M_E for the combination of two agents can be calculated using the formula $M_E = M_A + M_B(1 - M_A)$, where M_A and M_B are the observed mortalities for the two agents alone. Results from a chi-square test, $\chi^2 = (M_{AB} - M_E)^2/M_E$, where M_{AB} is the observed mortality for the combination, are compared to the chi-square table value for 1 df. If the calculated χ^2 value exceeds the table value, a non-additive effect between the two agents is suspected; if the difference $M_{AB} - M_E$ has a positive (negative) value, a significant interaction is considered synergistic (antagonistic).

ENTOMOPATHOGENIC NEMATODES

Several agents have been observed to synergistically enhance the infectivity of entomopathogenic nematodes to white grubs, the root-feeding larvae of scarabaeid beetles. *Paenibacillus* (= *Bacillus*) *popilliae*, the causative agent of milky disease in white grubs, facilitates nematode penetration through the midgut into the body cavity of the grubs. The slow establishment of milky disease in white grub field populations and the lack of in vitro production methods for the bacterium limit the feasibility of this combination.

Combination of the scarab-specific Buibui strain of *Bacillus thuringiensis* subsp. *japonensis* synergizes with nematodes and holds promise for control of scarab species with intermediate to high susceptibility to Buibui, but the commercialization of Buibui has been halted. The strongest synergism has been observed for combinations of the chloronicotinyl insecticide imidacloprid and nematodes. Exposure to imidacloprid facilitates nematode host attachment by inducing sluggishness and reducing defensive behaviors in the grubs. Because imidacloprid does not appear to compromise nematode recycling in grubs, this combination is not only promising for curative white grub control but could also play a role in augmentative and inoculative approaches to white grub management, especially of scarab species that are not very susceptible to nematodes.

Combinations of the pyrethroid insecticide tefluthrin and entomopathogenic nematodes resulted in synergistic corn rootworm mortality in the laboratory. Sublethal doses of the insecticide cause a paralytic and convulsive response in the insects that may render them more susceptible to nematodes.

ENTOMOPATHOGENIC FUNGI

The insecticide imidacloprid also synergizes with entomopathogenic fungi by temporarily reducing activity at sublethal doses. In subterranean termites, exposure to imidacloprid reduces removal of fungal conidia attached to the cuticle via social grooming, leading to increased susceptibility to various opportunistic and entomopathogenic fungi. In Citrus root weevil larvae, imidacloprid disrupts the normal conidial voidance accomplished through movement in the substrate, thus increasing fungal infection. Under field conditions, synergism was only observed occasionally, in part because of problems with leaching of the chemical. In the tarnished plant bug, synergism, at least with respect to speed of kill, was observed in the field but the mechanism of the interaction has not been studied.

ENTOMOPATHOGENIC VIRUSES

Optical brighteners, used as UV protectants in field applications of nucleopolyhedroviruses (NPV) against

Table 1 Examples of synergism between entomopathogenic microorganisms and other control agents or additives against insect pests

| Microorganism | Synergist | Host insect |
|-----------------------------------|--------------------------------|--|
| Nematodes ^a | | |
| <i>Hbac, Sgla</i> | <i>Paenibacillus popilliae</i> | <i>Cyclocephala hirta</i> |
| <i>Hbac, Sgla</i> | <i>Bacillus thuringiensis</i> | <i>Cyclocephala</i> spp. |
| <i>Hbac, Sgla, Hmeg, Hmar</i> | imidacloprid | <i>Cyclocephala</i> spp., <i>Popillia japonica</i> , <i>Exomala orientalis</i> |
| <i>Scar, Hbac</i> | tefluthrin | <i>Diabrotica virgifera virgifera</i> |
| Viruses ^b | | |
| Homologous ^c NPV | Tinopal LPW (M2R) | <i>Lymantria dispar</i> , <i>Spodoptera frugiperda</i> , <i>Pseudoplusia includens</i> |
| AfNPV | Blankophor P167 | <i>Helicoverpa zea</i> , <i>Spodoptera exigua</i> |
| Homologous NPV | Homologous GV | <i>Pseudaletia unipuncta</i> , <i>P. separata</i> , <i>Trichoplusia ni</i> , <i>Xestia c-nigrum</i> |
| <i>PuNPV</i> | <i>PsEPV</i> | <i>Pseudaletia separata</i> |
| <i>LdMNPV</i> | azadirachtin | <i>Lymantria dispar</i> |
| Fungi | | |
| <i>M. anisopliae, B. bassiana</i> | imidacloprid | <i>Diaprepes abbreviatus</i> , <i>Blatella germanica</i> , <i>Lygus lineolaris</i> , <i>Reticulitermes flavipes</i> |
| Bacteria | | |
| <i>Bacillus thuringiensis</i> | Inorganic salts | <i>Spodoptera littoralis</i> , <i>Agrotis ipsilon</i> , <i>Mamestra configurata</i> |
| | Amino acids | <i>S. littoralis</i> , <i>A. ipsilon</i> , <i>M. configurata</i> |
| | Organic acids | <i>S. littoralis</i> , <i>A. ipsilon</i> , <i>M. configurata</i> |
| | Protease inhibitors | <i>S. littoralis</i> , <i>A. ipsilon</i> , <i>M. configurata</i> |

^a*Heterorhabditis bacteriophora*, *H. marelatus*, *H. megidis*, *Steinernema glaseri*, *S. carpocapsae*.

^bNPV = Nucleopolyhedrovirus, GV = granulovirus, EPV = entomopoxvirus.

^cOriginating from the test insect.

lepidopteran pests, can concomitantly have a synergistic effect on mortality and speed of kill. The strongest synergism has been observed with the optical brightener Tinopal in larvae of the gypsy moth (virus: *LdMNPV*), the fall armyworm (virus: *SfMNPV*), and the soybean looper (virus: *PiMNPV*). The mechanism responsible for this interaction appears to be disruption of the sloughing of virus-infected primary target cells in the host midgut. The degree of interaction may depend on the physiological basis of resistance in a given host-virus combination. In ground-based applications against the gypsy moth, the addition of optical brightener has allowed a 10-fold reduction in virus concentrations. For aerial application, further research is needed to overcome uneconomically high optical brightener concentrations needed due to drop-let size requirement.

Other synergistic interactions with NPV have been observed in the laboratory only. The synergism between NPV and granuloviruses in several lepidopteran species is attributed to a metalloprotease, enhancer, in the granulovirus capsule that increases the NPV

virion permeability of the insect's peritrophic membrane. NPV infectivity is also synergized by the fusolin protein contained in the spheroid, spindle, and virion of entomopoxviruses; the mechanism of this interaction is unknown. The gypsy moth NPV has been synergized with the neem tree-derived triterpene azadirachtin in gypsy moth larvae, but the interaction could not be confirmed in field trials.

ENTOMOPATHOGENIC BACTERIA

Numerous studies have indicated synergistic effects between various strains or toxins of *Bacillus thuringiensis* and other control agents or formulation/spray additives. Much of this work has been conducted on artificial diets or leaf disc assays and needs verification under field conditions. The most promising synergists include salts (Ca²⁺-, K⁺-, Na⁺-, and Zn²⁺-salts), amino acids (e.g., arginine, glutamine, valine, proline), caffeine, inorganic acids (acetamine), trypsin inhibitors, and protein solubilizing reagents (EDTA, sodium

thioglycolate). The inorganic salts are inexpensive and have proven to increase crop yields in field trials by up to 5.7-fold (CaCO_3). Mechanisms responsible for synergistic interactions include aiding prototoxin solubilization by raising the gut pH (alkalis), increasing the permeability of the peritrophic membrane by abrasion (boric acid) or erosion (chitinase), increasing the permeability of the epithelial cells to the toxin (detergents), or increasing the concentration of cofactors (metal ions) of enzymes that cleave prototoxins into active toxins.

OUTLOOK

The ultimate goal of studies on synergistic interaction with microorganisms is the development of environmentally sound and economically feasible alternatives to hazardous pesticides. While several of the described synergistic actions have been confirmed under field conditions, none of them has so far found widespread application. Generally, the degree of synergism tends to be stronger in pests that are difficult to control with the microorganisms alone. In these cases, synergism can improve the economy of the applications by decreasing the effective dosage of the individual control agents. Although some of these combinations could become highly effective with some further fine-tuning, economics and user friendliness of formulations and application techniques will ultimately determine their spread.

[See also Biological pest controls, insects and mites; Biological pest controls, fungal control of pest; nematode control of pests; pesticides, auxiliaries; pesticides, biopesticides.]

BIBLIOGRAPHY

Boucias, D.G.; Stokes, C.; Storey, G.; Pendland, J. Effect of imidacloprid on the termite, *Reticulitermes flavipes* and

its interaction with insect pathogens. *Pflanzenschutz-Nachr. Bayer* **1996**, *49*, 103–150.

Burges, H.D.; Jones, K.A. Formulation of Bacteria, Viruses and Protozoa to Control Insects. In *Formulation of Microbial Biopesticides: Beneficial Microorganism, Nematodes and Seed Treatments*; Burges, H.D., Ed.; Kluwer: Dordrecht, Netherlands, 1998; 33–127.

Jaques, R.P.; Morris, O.N. Compatibility with Other Methods of Pest Control and with Different Crops. In *Microbial Control of Pests and Plant Diseases 1970–1980*; Burges, H.D., Ed.; Academic Press: London, England, 1981; 695–715.

Koppenhöfer, A.M.; Choo, H.Y.; Kaya, H.K.; Lee, D.W.; Gelernter, W. Improved field and greenhouse efficacy with a combination of entomopathogenic nematode and *Bacillus thuringiensis* against scarab grubs. *Biol. Control* **1999**, *14*, 37–44.

Koppenhöfer, A.M.; Grewal, P.S.; Kaya, H.K. Synergism of imidacloprid and entomopathogenic nematodes against white grubs: the mechanism. *Entomol. Exp. Appl.* **2000**, *94*, 283–293.

Nishimatsu, T.; Jackson, J.J. Interaction of insecticides, entomopathogenic nematodes, and larvae of the western corn rootworm (Coleoptera; Chrysomelidae). *J. Econ. Entomol.* **1998**, *91*, 410–418.

Quintela, E.D.; McCoy, C.W. Synergistic effect of two entomopathogenic fungi and imidacloprid on the behavior and survival of larvae of *Diaprepes abbreviatus* (Coleoptera: Curculionidae) in soil. *J. Econ. Entomol.* **1998**, *91*, 110–122.

Thorpe, K. W.; Cook, S. P.; Webb, R. E.; Podgwaite, J. D.; Reardon, R. C. Aerial application of the viral enhancer Blankophor BBH with reduced rates of gypsy moth (Lepidoptera: Lymantriidae) nucleopolyhedrovirus. *Biol. Control* **1999**, *16*, 209–216.

Thurston, G.S.; Kaya, H.K.; Gaugler, R. Characterizing the enhanced susceptibility of milky disease-infected scarabaeid grubs to entomopathogenic nematodes. *Biol. Control* **1994**, *4*, 67–73.

Washburn, J.O.; Kirkpatrick, B.A.; Haas-Stapleton, E.; Volkman, L.E. Evidence that the stilbene-derived optical brightener M2R enhances *Autographa californica* M nucleopolyhedrovirus infection of *Trichoplusia ni* and *Heliothis virescens* by preventing sloughing of infect mid-gut epithelial cells. *Biol. Control* **1998**, *11*, 58–69.

Systematics and Biological Pest Control

Amy Y. Rossman
Michael Schauff

*Systematic Botany and Mycology Laboratory, United States Department of Agriculture,
Agricultural Research Service, Beltsville, Maryland, U.S.A.*

INTRODUCTION

Systematics is the science that identifies and characterizes all organisms, i.e., the study of biological diversity. Systematic knowledge is essential for discovering biological control agents and determining how these organisms will function when introduced into new habitats. Numerous insects, fungi, and other microorganisms exist that hold great potential for controlling serious pests including diseases, but most are virtually undiscovered, much less described, characterized, and tested for their potential in controlling deleterious organisms. If the biological control potential of insects, fungi, and other microorganisms were harnessed, the use of chemicals that pollute our food and the environment would be considerably reduced. This can happen safely and effectively only if species used in biological control are accurately identified and thoroughly characterized, with knowledge obtained through systematics research.

The major kinds of organisms used most successfully in biological control are insects and fungi, although bacteria and viruses have also proven useful. These are the “megadiverse” groups of organisms about which relatively little is known considering the enormity of their biological diversity. With the increased exploration for these speciose groups of organisms, many new species with biological control potential are being discovered. In addition to testing for biological control properties, these species must also be described, named, and accurately characterized prior to their introduction as biological control agents.

WHY IS SYSTEMATICS IMPORTANT TO BIOLOGICAL PEST CONTROL?

Systematics is uniquely positioned to support biological control programs by providing critical information on relationships among species, distributional information, host associations, and other biological data. Authoritative identification of both host and beneficial organisms allows access to biological information as well as entry into relevant literature. Biocontrol programs have been delayed, suffered from diminished

success, or failed outright because of lack of systematic information.^[1] When accurate systematic knowledge is erroneous or incomplete for a potential control agent, crucial biological information may not be found because it is hidden under other scientific names. If a biological control agent is insufficiently characterized, once released, it may be impossible to distinguish the released agent from native organisms; thus the effectiveness of the introduced species cannot be documented. Time and resources may be wasted such as importing species already present in the ecosystem or rearing and releasing inappropriate natural enemies. Two classic examples of the kind of problem encountered because of lack of systematic input into biocontrol programs involve the Chinese wax scale (*Ceroplastes sinensis*) and a parasitic wasp. In this case, the lack of a good systematic knowledge led workers to search for natural enemies in the wrong part of the world. In the second case, about 4 million insects were reared and released only to have discovered that these were the wrong species of parasitoid (*Encarsia fasciata*).^[2,3]

METHODS USED IN SYSTEMATICS OF ORGANISMS USED IN BIOLOGICAL PEST CONTROL

Methods used in obtaining systematic knowledge of biological control agents vary depending on the kind of organism involved. Fungi are characterized primarily using the morphology of microscopic structures;^[4] thus it is necessary to use both stereoscopic and compound microscopes, often supplemented by the scanning electron microscope. Most fungi must also be grown on defined media in Petri plates at specified temperatures.^[5] Because identification and characterization of microfungi are difficult, some species can only be determined by sequencing specific gene regions such as the internally transcribed spacer (ITS) region of the nuclear ribosomal DNA and comparing the unknown sequence with known sequences. This approach is limited by the availability and accuracy of the sequences in GenBank. The literature on the systematics of a specific group of organisms must be

consulted to identify and characterize a potential biological control agent. Increasingly, these resources are available on the Internet. Insects and mites are identified in much the same way as mentioned for fungi. Morphological characters are assessed using a combination of light and scanning electron microscopy. For problems involving cryptic species, geographic races or biotypes, and, increasingly, even higher-level relationships, molecular data are becoming important. Identification aids are increasingly available to non-specialists as interactive keys and digital photographs on the Internet. In particular, expert systems using software programs such as LucID allow identifications based on a few diagnostic characters.

SYSTEMATICS OF MAJOR GROUPS OF ORGANISMS USED IN BIOLOGICAL PEST CONTROL: FUNGI

Fungi are used primarily in the biological control of weeds, insects, and fungal plant pathogens. The fungi most commonly used to control weeds include mitosporic ascomycetes such as *Colletotrichum*, *Phoma*, and *Phomopsis* in addition to obligate parasites such as the rust fungi (Uredinales, Fig. 1) and Oomycetes such as *Phytophthora*. General references on mitotic ascomycetes^[6-8] can be used to identify a fungus to genus; however, specialized literature is required for identification to species.^[9] Identification of rust fungi is based primarily on plant host.^[10]

Fungi have been successfully used to control insects and nematodes especially in greenhouse situations. The major groups of insect-associated fungi are mitosporic hypocrealean ascomycetes and obligate parasites in the Entomophthorales.^[11] Fungi associated with nematodes are extremely diverse,^[12] although most research has centered on the hypocrealean ascomycetes *Pochonia chlamydosporia* (formerly *Verticillium chlamydosporium*, unrelated to the plant pathogens).

Many plant diseases can be controlled using fungi such as mitosporic ascomycetes. Identification of

mitosporic ascomycetes is difficult as mentioned above and may best be accomplished through sequencing. Accurate species identification is crucial as exemplified by the confusion between *Trichoderma harzianum*, used in the control of many plant diseases, and *Trichoderma aggressivum*, initially identified as *T. harzianum*, causing green mold in cultivated mushrooms beds.^[13] Basidiomycetes such as *Rhizoctonia* and Oomycetes such as *Pythium* have also been used to control fungal diseases.

SYSTEMATICS OF MAJOR GROUPS OF ORGANISMS USED IN BIOLOGICAL PEST CONTROL: INSECTS AND MITES

A wide array of insect and mite groups has been used for control of pests and weeds.^[14,15] The importation of the Vedalia beetle, a predatory lady beetle (Coccinellidae), into California in the 1880s to control the cottony cushion scale was the first widely documented successful biological control effort. Parasitic wasps (Hymenoptera), such as the chalcid wasps (Chalcidoidea), braconid wasps (Braconidae), and ichneumon wasps (Ichneumonidae), have been used successfully in pest control programs. Among the true flies, the parasitic Tachinidae have been used against a number of caterpillars (Lepidoptera) and other groups. Predatory groups include lady beetles (Coccinellidae, Fig. 2), assassin bugs (Reduviidae), lacewings (Chrysopidae), flower flies (Syrphidae), and predatory mites (Phytoseidae). Plant-feeding insects and mites are used to combat invasive weeds.^[16] Among the major insect groups used are the leaf beetles (Chrysomelidae), weevils (Curculionidae), underwing

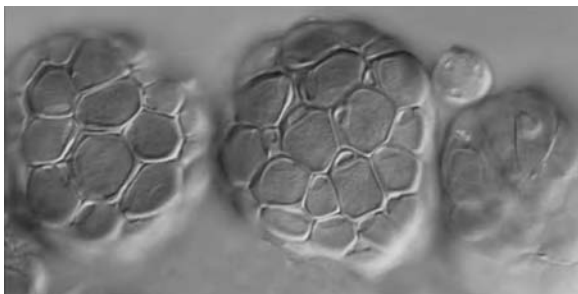


Fig. 1 Teliospores of rust *Ravenelia heironymi* on *Acacia* spp. (photograph by Jose Hernández).



Fig. 2 A lady beetle (Coccinellidae) feeding on an aphid (photo by Scott Bauer).

moths (Noctuidae), gall-making flies (Cecidomyiidae), and psyllids (Psyllidae).

CHALLENGES IN SYSTEMATICS AND BIOLOGICAL PEST CONTROL

With the application of molecular techniques, systematic knowledge has progressed rapidly in the last decade especially in biologically diverse groups such as fungi and insects, yet their accurate identification and characterization remain difficult. Often, the potential biological agent is an undiscovered species that must be carefully described and characterized in relation to known species.^[17] At a time when the vast diversity of fungi and insects is being discovered, the number of scientists with expertise in systematics of these organisms is declining.^[18] Once this systematics expertise declines below a critical level, it may be difficult to make progress in applied research areas such as development of biological pest control that depend on systematic knowledge for success.

FUTURE PROSPECTS

Increasingly, systematics resources for the identification and characterization of organisms useful in biological pest control are available on the Internet. These include well-illustrated, on-line identification systems aimed at non-specialists such as for the mitotic ascomycetes *Trichoderma* and the rust genus *Ravenelia*.^[19] In addition, annotated lists of species on hosts and literature useful in identifying biological pest control organisms are readily available. Molecular systematics especially the exponentially increasing number of organisms represented by sequences in GenBank provides tools for accurately determining the identification and phylogeny of fungi and insects.

REFERENCES

1. Schauff, M.E.; LaSalle, J. The relevance of systematics to biological control: protecting the investment in research. In *Pest Management—Future Challenges*, Proceedings of the 6th Australian Applied Entomological Conference, Brisbane, Australia, 1998, Vol. 1. 425–436.
2. Qin, T.; Gullan, P.J.; Beattie, G.A.C.; Trueman, J.W.H.; Cranston, P.S.; Fletcher, M.J.; Sands, D.P.A. The

- current distribution and geographical origin of the scale insect pest *Ceroplastes sinensis* (Hemiptera: Coccoidea). *Bull. Entomol. Res.* **1994**, *84* (4), 541–549.
3. Rosen, D.; DeBach, P. Use of scale-insect parasites in Coccoidea systematics. *Res. Div. Bull.-Va. Polytech. Inst. State Univ.* **1977**, *127* (1), 5–21.
4. Rossman, A.Y.; Tulloss, R.E.; O'Dell, T.; Thorn, R.G. *Protocols for an All Taxa Biodiversity Inventory of Fungi in a Costa Rican Conservation Area*; Parkway Publishers: Boone, NC, 1998.
5. Malloch, D. *Moulds. Their Isolation, Cultivation and Identification*; University of Toronto: Toronto, Ontario, 1981.
6. Carmichael, J.W.; Kendrick, W.B.; Connors, I.L.; Sigler, L. *Genera of Hyphomycetes*; University of Alberta: Edmonton, Alberta, 1980.
7. Nag Raj, T.R. *Coelomycetous Anamorphs with Appendage-Bearing Conidia*; Mycologue Publ.: Waterloo, Ontario, 1993.
8. Sutton, B.C. *The Coelomycetes*; Commonwealth Mycological Institute: Kew, Surrey, England, 1980.
9. <http://nt.ars-grin.gov/fungaldatabases/literature/litframe.cfm> (accessed October 2003).
10. Cummins, G.B.; Hiratsuka, Y. *Illustrated Genera of Rust Fungi*, 3rd Ed.; American Phytopathological Society: St. Paul, MN, 2003.
11. Samson, R.A.; Evans, H.C.; Latge, J.-P. *Atlas of Entomopathogenic Fungi*; Springer-Verlag: Berlin, 1988.
12. Carris, L.M.; Glawe, D.A. Fungi colonizing cysts of *Heterodera glycines*. *Univ. Ill. Urbana-Champaign Bull.* **1989**, 785.
13. Samuels, G.J.; Dodd, S.L.; Gams, W.; Castlebury, L.A.; Petrini, O. *Trichoderma* species associated with the green mold epidemic of commercially grown *Agaricus bisporus*. *Mycologia* **2002**, *94* (1), 146–170.
14. Clausen, C.P. Introduced parasites and predators of arthropod pests and weeds. *USDA Agric. Handbook* **1978**, 480.
15. Quicke, D.L.J. *Parasitic Wasps*; Chapman and Hall: London, 1997.
16. Julien, M.H. *Biological Control of Weeds: A World Catalogue of Agents and Their Target Weeds*, 3rd Ed.; CABI Publishing: Wallingford, United Kingdom, 1997.
17. Samuels, G.J.; Pardo-Schultheiss, R.A.; Hebbar, K.P.; Lumsden, R.D.; Bastos, C.N.; Costa, J.C.; Bezerra, J.L. *Trichoderma stromaticum* sp. nov., a parasite of the cacao witches broom pathogen. *Mycol. Res.* **2000**, *104* (6), 760–764.
18. House of Lords. *What on Earth? The Threat to the Science Underpinning Conservation. HL Paper 118(i)*; Stationary Office: London, 2002.
19. <http://nt.ars-grin.gov/taxadescriptions/keys/> (accessed October 2003).

Systemic Insecticides

Nick C. Toscano

Department of Entomology, University of California, Riverside, California, U.S.A.

Nilima Prabhaker

Western Cotton Res Lab, University of California, Riverside, Phoenix, Arizona, U.S.A.

INTRODUCTION

Systemic insecticides are represented in various classes of insecticides. They are uniquely characterized by their systemic distribution within organisms treated to defend against pestiferous insects and other arthropods. Although the principal use of systemic insecticides is in crop protection, a few select compounds with low mammalian toxicity can also be used in a medical and veterinary context to counter certain pests such as fleas and ticks as well as by homeowners for household pests. Distribution of systemic insecticides within plants occurs by absorption either through plant roots following soil application or foliage and stems following spray application. The advantages of using soil-applied systemic insecticides are greater selectivity and increased flexibility in pest management programs that seek to conserve beneficial insects. But because of their systemic occurrence within crop plants grown for human consumption, caution is required to avoid contamination. Newer, systemic insecticides are available that are safer and more effective than compounds developed decades earlier.

CHARACTERISTICS OF SYSTEMIC INSECTICIDES

Systemic insecticides are toxicants that are applied to the roots or foliage of growing plants where they are absorbed and translocated throughout the plant. The application of a systemic insecticide, whether above ground to the foliage and stems or below the ground surface to the root zone, depends on the physical properties of each compound. Many organophosphorous compounds have both contact and systemic properties and in some cases are formulated specifically for either usage. Conceptually, systemic insecticides are generally thought of as soil-applied compounds taken up by roots and distributed throughout the plant despite the fact that many are primarily applied foliarly.

Systemic insecticides can also be injected directly into the trunks of trees without making any contact

with the environment. Trunk injection of systemics allows an efficient delivery system of the material where they are dispersed fairly fast through the vascular system to all parts of the tree. This technique is frequently being used in urban settings as an alternative to spray applications.

Most conventional insecticides work by directly contacting with insects either at the time of a spray application or as residues on plant surfaces while walking or alighting on the treated surface. Some of these foliar-applied contact insecticides are able to penetrate the cuticle of plants and move to sub-cuticular tissues by a process called translaminar mobility. The limited intercellular movement or transfer across membranes that occurs with translaminar-active compounds should not be confused with translocation of a systemic insecticide throughout a plant by transport in the vascular system.

Modern synthetic insecticides are foremost identified by the chemical class to which they belong, e.g. organophosphates, carbamates, pyrethroids, organochlorines, etc. Within each class, tremendous diversity can occur in the overall structure of the compounds built around the unique region, or moiety, that signifies a particular class. But it is the entire molecular structure of a compound that confers a set of biophysical properties which determines the compound's relative toxicity at a target site within a pest organism, how well it is transported to that target site, its vulnerability to metabolic degradation, its stability in the environment following application, how it interacts with a plant on contact, and numerous other characteristics that determine how any one compound is most effective to combat pests.

One critical physical requirement for systemic insecticides is that they be sufficiently soluble in water to permit absorption by plant roots as well as systemic translocation within the plant's vascular system. Compounds within any chemical class that are somewhat water-soluble are potential candidates for systemic application, but many other factors determine whether an insecticide will protect plants as a systemic or a contact insecticide. The degree of water solubility influences the nature of the protection afforded to plants.

For example, more soluble compounds, such as the organophosphate insecticide acephate (~65% solubility), typically will have fairly short-term activity in plants because of their tendency to disperse in soil with the movement of water out of the root zone. Aldicarb, a carbamate, has much lower water solubility (~0.6%) and is less mobile in soil than acephate and therefore available for root absorption over a longer period. The greater intrinsic toxicity of aldicarb relative to acephate allows it to be highly effective and longer lasting at protecting plants systemically, even at lower concentrations. The specific characteristics of any one compound, systemic or otherwise, must be taken into consideration when trying to optimize pest management strategies.

AGRICULTURAL USES AND IMPORTANCE

Insects or mites feeding on the sap or tissues of treated plants are exposed to systemic insecticides primarily through ingestion,^[1] although a foliar-applied systemic would also have contact activity. The target pests for systemic insecticides tend to be smaller-sized and include aphids, lace bugs, mealybugs, scales, spider mites and whiteflies. Certain systemic insecticides also control some chewing insects, notably the Colorado potato beetle (*Leptinotarsa decemlineata* (Say)). Great success in management of this notoriously difficult pest of potatoes has been achieved with the recently introduced imidacloprid, a neonicotinoid compound formulated for use both as a root zone and foliar-applied systemic insecticide. First commercialized in 1991, the superb attributes of imidacloprid including its systemic activity and high toxicity against certain insects, but very low toxicity against mammals, birds and fish, propelled this compound to number 1 in worldwide insecticide sales by 1996.

The unique attributes of systemic insecticides add versatility to pest management by increasing chemical control options available to pest managers. In making treatment decisions, not only are there more insecticides to select from, but the number of strategies for treating particular pest situations is also increased. For example, in the case where emergent seedlings are vulnerable to insect attack in a freshly planted field, access with a tractor sprayer could be restricted until the field had firmed following the post-sowing irrigation. Meanwhile, the crop stand could be severely impacted unless a pre-emergent application of a systemic insecticide was made prior to irrigation. Another example involves controlling insects that live within plant tissue or in concealed regions of a plant that normally are not accessible by contact insecticides. In such cases, systemic insecticides are invaluable for protecting plants against specialized pests. Similarly,

to control borers and scale on trees, trunk injections or implants of systemic insecticides are efficient. However, injections of systemics are less efficient in evergreens than in deciduous trees because of closure of injection holes as a result of copious pitch production.^[2] Another case involves sustained attacks by a pest population immigrating into a field from surrounding fields of an earlier planted crop. Whereas contact insecticide treatments may be very effective at combating significant colonization, costly repeated applications might be necessary to avoid damaging infestations. A long-lasting systemic insecticide, however, would provide continuous protection and help prevent secondary disruptions through repeated spray applications.

BENEFITS AND COSTS OF SYSTEMIC INSECTICIDE USE

As with any agrochemical, the potential benefits of systemic insecticides must be weighed against the short and long-term costs of their use. The regulatory processes in most developed nations provide a stringent set of bio- and eco-toxicity standards for registration and reregistration of new and existing commercial products, respectively. The central issue concerns whether the level of impact on human health and non-target organisms in the environment is acceptable based on the specific use of a product. Systemic insecticides have some unique properties that provide advantages from a pest management standpoint, but also require precautions in terms of how they are used in the environment.

Benefits

A distinct advantage of systemic insecticides is that they are ecologically more selective than insecticides applied by air or ground spray. Loss of natural enemies, resurgence of a pest population and outbreak of secondary pests (when it occurs) are the undesirable outcomes of the use of broad-spectrum, foliar insecticides. Soil-applied systemic insecticides largely circumvent the indiscriminate kill of pest and beneficial insects alike by being taken up by the roots and distributed systemically throughout the plant. Preferentially, toxic exposure should be limited to those insects and mites that feed upon the plant while leaving beneficial insects unharmed and intact for integrated control. This ideal is largely achieved when a systemic insecticide is delivered to the root zone without exposing the above-ground fauna, pest or beneficial. But once the systemic is taken up and distributed to all parts of the plant, only those insects feeding on the plant

Table 1 General comparisons between soil-applied systemic insecticides and foliar-applied contact insecticides

| Category | Soil-applied systemic insecticides | Foliar-applied contact insecticide |
|--------------------|---|---|
| Specificity | Much reduced exposure of non-target organisms, generally only plant feeders are exposed. | Indiscriminate contact of pest and beneficial organisms. |
| IPM Compatibility | More easily integrated, better conservation of beneficials. | Often disruptive to IPM because of reduced natural mortality of pests. |
| Coverage | More complete coverage through systemic distribution; better protection. | Limited underleaf and under canopy coverage; more untreated escapes. |
| Protection window | Usually longer residual activity; fewer applications required. | Often short residual activity limited by UV degradation or volatilization. |
| Specialized pests | Effective against both internal and external plant feeders. | Internal feeders, e.g. leafminers, or hidden feeders, e.g. aphids and thrips, not contacted by foliar sprays. ^{1a} |
| Virus transmission | Reduced transmission of phloem-limited viruses. | No direct impact on virus transmission other than vector control. |
| Environment | No spray drift; less hazard to non-target organisms including birds and fish; potentially increased risk of ground water contamination. | Increased risk of exposure to organisms outside of crop by spray drift. |

^aMay not apply to foliar sprays having translaminar activity.

presumably are at risk of being poisoned by ingesting the systemic insecticide. However, indirect exposure of beneficials to toxicants as a result of feeding on contaminated prey has not been studied extensively. This indirect exposure can result in altered behavior and reduced longevity of the beneficials. Reduced performance of beneficials in parasiting and preying on hosts when exposed to sublethal amounts of systemic insecticides has been observed.^[3]

In addition to the selectivity advantages that soil-systemic insecticides have in integrated pest management, other features also make them the best choice in various pest control situations (Table 1). For example, systemics can be used to control virus vectors that transmit plant diseases and potentially reduce the spread of diseases. Moreover, transmission of phloem or xylem limited plant pathogens can be prevented by a systemic because mortality of the vector occurs before it attains the specific tissue required for transmission. Imidacloprid increased the mortality of beet leafhopper and reduced transmission of beet curly top virus in cucumbers compared with foliar sprays of dimethoate,^[4] and potato leafroll virus transmission by aphids was reduced in potatoes.^[5]

Control of insect vectors and the diseases they transmit, and targeted pest control in general, are often greatly prolonged with a systemic insecticide treatment. Long residual availability in the soil and concomitant activity in plants preclude the need for repeated treatments of shorter residual contact insecticides making it economically beneficial for growers.

The vascular-distributed systemic insecticide provides whole plant protection whereas spray contact insecticides are frequently limited by incomplete coverage of plants.

Costs

Systemic insecticides with prolonged uptake and activity within plants have also been assumed to be more vulnerable to resistance development in pest populations. The combination of a longer exposure period and more uniform distribution within plants theoretically intensifies selection pressure. In some instances, however, fewer applications and more complete kill could counter the tendency towards faster resistance. There seems to be no clear-cut evidence that systemic insecticides have been any more likely to develop resistance faster than contact insecticides.

Another potential concern of soil applications is their fate in the soil environment and the possibility of contamination of runoff water and/or groundwater. This depends on the properties of individual compounds such as the solubility and susceptibility to degradation and the soil characteristics. For example, the use of aldicarb on Florida citrus is severely restricted because of past problems of rapid leaching from sandy and acidic soils and contamination of drinking water wells. Other states also restrict the use of aldicarb over groundwater concerns as well as problems

with higher-than-acceptable residues in potatoes and other food crops.

OUTLOOK FOR SYSTEMIC INSECTICIDES

Increasing emphasis is being placed on insecticides with selective activity which provide more precise pest control. Soil-applied systemic insecticides are selectively active against those insects that feed on a treated plant, thus helping to conserve beneficial insects through reduced exposure. In addition to imidacloprid, other neonicotinoid compounds under development all show systemic properties that provide long-lasting pest control. Other attributes of neonicotinoids include low toxicity to vertebrates and comparatively low use rates making this an increasingly important class of insecticides.

REFERENCES

1. Bennett, S.H. Preliminary experiments with systemic insecticides. *Ann. Appl. Biol.* **1959**, *36*, 160–163.
2. Tattar, T.A. Microinjection: small package, big impact. *Landsc. Manage.* **1999**, *32*, 28–30.
3. Stapel, J.O.; Cortesero, A.M.; Lewis, W.J. Disruptive sublethal effects of insecticides on biological control: Altered foraging ability and life span of a parasitoid after feeding on extrafloral nectar of cotton treated with systemic insecticides. *Biol. Control* **2000**, *17*, 243–249.
4. Wang, H.; Gurusinghe, P.de A.; Falk, B.W. Systemic insecticides and plant age affect beet curly top virus transmission to selected host plants. *Plant Dis.* **1999**, *83*, 351–355.
5. James, S.R.; Reed, G.L. Using admire and Temik to control aphids on russet potatoes. In *Central Oregon Agricultural Research Center, Annual Report; Spec. Rep.*; 1995; Vol. 955, 46–52.

Tea Diseases: Ecology and Control

N. Muraleedharan

U.I. Baby

UPASI Tea Research Foundation, Valparai, Coimbatore, Tamil Nadu, India

INTRODUCTION

The tea plant of commerce [*Camellia sinensis* (L) O. Kuntze] is grown in more than 50 countries lying between 43°N and 42°S latitudes and from sea level to 2300 m above mean sea level. These plants prefer warm humid climatic conditions, well-distributed rainfall, and long sunshine hours. These conditions are conducive for the growth of many pests and pathogens and further, the monoculture habitat provides a stable microclimate for their easy transmission and establishment. Four hundred pathogens have been reported from tea,^[1] of which the majority are fungi, and a few are bacteria, viruses, and algae. Common names of tea diseases and the pathogen associated have been documented recently.^[2]

Root Diseases

Primary root diseases generally occur in areas planted with tea after clearing virgin forest or where shade trees have been removed without ring barking. These pathogens are true parasites and are transmitted mainly through root contact and through spores. In the case of secondary root diseases the pathogens infect the plants which are weakened by certain predisposing factors. The major root rots in Southeast Asia are red root, brown root, and charcoal stump rot. The predominant root diseases in Africa are root splitting [*Armillaria mellea* (Fr.) Vahl] and charcoal stump rot [*Ustilina deusta* (Fr.) Petrak]. In Japan it is white root rot [*Rosellinia necatrix* (Hartig) Berl.]. In southern China seedling root rot [*Sclerotium rolfsii* (Sacc.) Curzi], red root [*Poria hypolateritia* (Berk.) Cooke], and violet root rot (*Helicobasidium purpureum* Pat.) are the major diseases. In northern India, brown root [*Fomes lamaoensis* (Murr.) Sacc. and Trott.], charcoal stump rot [*Ustilina zonata* (Lev.) Sacc.], red root and black root rot (*Rosellinia arcuata* Petch) are the principal diseases, while in southern India red root, brown root (*Fomes noxius* Corner), black root, charcoal stump rot, violet root rot (*Sphaerostilbe repens* B. and Br.), and diplodia root disease (*Botryodiplodia theobromae* Pat.) are the most common.

Charcoal stump rot is widespread in India, Sri Lanka, and Indonesia. Two species *U. deusta* (Fr.) Petrak and

U. zonata (Lev.) Sacc. are associated with the disease and the latter is reported from southern India. The disease is common in areas affected by lightning. Violet root rot (*S. repens* B. and Br.) is associated with water-logging conditions of the soil and is common on bushes growing near the swamp. The disease also occurs in hard and poorly aerated soil. Another important secondary root disease is diplodia disease (*Botryodiplodia theobromae* Pat.). Lack of starch reserves, general debility due to poor nutrition, untimely pruning, continuous hard plucking, repeated incidence of pests and leaf diseases are considered as predisposing factors of the disease. Other common root diseases are xylaria root disease (*Xylaria* sp.), purple root rot [*Helicobasidium compactum* (Boedijn) Boedijn], tarry root rot [*Hypoxylon asarcodes* (Theiss.) Mill.], and root rot (*Cylindrocarpon tenue* Bugnicourt).

Stem Diseases

A number of parasitic and saprophytic organisms enter through the prune cuts and wounds on frames, resulting in various stem diseases. Wood rot (*Hypoxylon*) is the most serious stem disease in all tea-growing countries. The affected branches and stem become dry, light, brittle, and nonfunctional, resulting in progressive debilitation of the bushes with consequent loss of crop and ultimate death of the bushes. The predominant species in India is *Hypoxylon serpens* (Pers. Fr.) J. Kickx, and other species involved are *Hypoxylon nummularium* Bull. Fr. and *Hypoxylon vestitum* Petch. In Africa, besides *H. serpens*, *Hypoxylon investiens* has also been observed. Collar canker (*Phomopsis theae* Petch) and branch canker (*Macrophoma theicola* Petch) are the other important diseases that could inflict heavy economic losses. Collar canker is more serious and occasionally badly damaged fields require complete replanting. Thorny stem blight caused by *Tunstallia aculeata* (Petch) Agnihotrudu is of concern in India, Sri Lanka, Columbia, and Indonesia. There are many other stem diseases which are of minor importance. These include pink disease (*Corticium salmonicolor* Berk & Br.), velvet blight (*Septobasidium bogoriense* Pat.), stem canker (*Poria hypobrunnea* Petch), branch canker (*Nectria haematococca* Berk & Br.), die back [*Leptothyrium theae* Petch, *Nectria*

cinnabarina (Tode: Fr.) Fr.], and thread blight [*Marasmius tenuissimus* (Junghuhn) Singer].

Leaf Diseases

Foliar diseases, especially those affecting young shoots, have a direct effect on the crop. On the other hand, those affecting mature leaves have an indirect effect on the crop by influencing photosynthesis. Among the foliar diseases, blister blight incited by *Exobasidium vexans* Masee is the most important one. Blister blight attacks only the succulent leaves and stem of the harvestable shoots leading to heavy crop loss. The disease spreads through wind-borne basidiospores. The entire life cycle of *E. vexans* is completed in 11–28 days under conducive climatic conditions. A monthly temperature of 12–20°C, moderate rainfall, high relative humidity (>80%), as well as availability of tender shoots are congenial for its occurrence.

Black rot is a serious disease in northeastern India and in some of the inadequately ventilated sections of tea gardens in southern India. Two species of the fungus *Corticium*, viz., *Corticium invisum* Petch. and *Corticium theae* Barnard, are involved with the disease. The fungus persists on the same bush and spreads to neighboring bushes under conducive conditions. The disease debilitates the bushes leading to severe crop loss.

Diseases of mature leaves such as grey blight [*Pestalotiopsis theae* (Sawada) Steyaert], brown blight [*Colletotrichum camelliae* Masee], anthracnose [*Colletotrichum theae-sinensis* (Miyake) Yamamoto], and net blister blight (*Exobasidium reticulatum* Ito & Sawada) reduce crop production by prolonging the banji (dormant shoots) period. Anthracnose and net blister blight are significant in China, Japan, and Taiwan, while gray blight and brown blight are common in almost all tea-growing countries. White scab (*Elsinoe leucospila* Bitancourt & Jenkins), white spot (*Phyllosticta theaeifolia* Hara), bird's eye spot [*Cercospora theae* (Cavara) Breda de Haan, *Pseudocercospora theae* (Cavara) Deighton], brown spot (*Calonectria colhounii* Peerally), sooty mold (*Meliola camelliae* and *Capnodium theae* Boedijn) are other common foliar diseases.

Red rust is an algal disease affecting mature leaves and young stem. The disease is caused by three species of *Cephaleuros*, viz., *Cephaleuros parasiticus* Karsten, *Cephaleuros virescens* Kunze, and *Cephaleuros mycoidea* Karst. It is fairly widespread in India, Sri Lanka, Indonesia, and Malaysia. The alga invades weak and debilitated tea plants. It spreads through wind- and water-disseminated sporangia.

Bacterial and viral diseases are of negligible importance in tea. The bacterial diseases recorded are shoot

blight [*Pseudomonas syringae* pv. *theae* (Hori) Young, Dye & Wilkie] and bacterial leaf spot and canker (*Xanthomonas campestris* pv. *theicola* Uehara, Arai, Nonaka & Sano, *Xanthomonas gortencovianum* Daneliya & Tsilosani). Only one viral disease, the phloem necrosis (*Camellia virus 1*) has been recorded so far.

Diseases of Nursery Plants

Stalk rot (*Pestalotia theae*, *Colletotrichum camelliae*), damping-off (*Cylindrocladium*, *Pythium* spp.), root rot (*Cylindrocladium*, *Pythium*, *Fusarium* spp.), collar rot (*Rhizoctonia solani*), blister blight (*Exobasidium vexans*), and leaf spot (*Cercospora theae*) are common diseases affecting tea plants in the nursery.

DISEASE MANAGEMENT

Tea diseases cause considerable crop loss by debilitating the bushes or killing them. The crop loss depends on the nature of the disease and the plant part affected. On a global basis, the crop loss due to diseases varies from 10% to 15%, indicating the need for adopting proper disease management strategies.

Disease Resistance

A wide variety of clones are available for commercial planting and these show varying levels of resistance to different diseases. There are specific clones which are tolerant to branch and stem canker, blister blight, anthracnose, grey blight, and brown blight. However, there are no clones with a high degree of resistance to root diseases. Though conventional breeding and biotechnological approaches are being attempted to develop resistant varieties to various diseases, this may have only limited significance in perennial crops like tea as resistance breaks down over a period of time.

Cultural Control

Certain cultural operations can prevent the development of pathogenic organisms in new areas. Occurrence of primary root diseases can be prevented by removing all potential sources of infection at the time of clearing jungle land, removing mature shade trees, or uprooting old tea plants for replanting. Ring barking of trees depletes root carbohydrate reserves, which encourage the colonization of saprophytic microorganisms on roots, after felling. This operation is widely practiced in India, Sri Lanka, and Indonesia.^[3] Isolation of diseased patches by taking trenches of

120 cm depth and 45 cm width, surrounding patch, prevent the spread of primary root diseases. While taking trenches, one or two rows of apparently healthy bushes also have to be included in the patch and all of them should be dug out and disposed off.^[4] The area should not be replanted without adequate soil treatments. In the case of charcoal stump rot, the affected bushes have to be uprooted and the area then put under rehabilitation. Black root disease spreads through the mulch and decaying organic matter. Hence, in its management, the organic matter at least 50 ft around the focus of infection should be removed, burnt, and the soil kept bare. Soil rehabilitation by growing nonhosts, like Guatemala grass (*Tripsacum laxum*) or thornless mimosa (*Mimosa invisa*) for about 2 yr helps to deplete the inoculum in soil and also to improve the physical structure and organic matter status of the soil. The basic concept of this approach is that primary root pathogens cannot persist for long in the absence of a host. In the case of secondary root diseases the control depends on the factor(s) predisposing the plants to infection by the fungus concerned, rather than directly preventing the fungal invasion.

Manipulation of certain cultural practices such as pruning, plucking, shade regulation, and weed control can reduce the incidence of many diseases. Continuous harvesting with shears debilitates the bushes and predisposes them to various diseases like diplodia root disease, thorny stem blight, grey blight, and red rust. Improvement of bush health through balanced nutrition and avoiding too much stress on bushes prevent their occurrence. Cultural operations like pegging, planting in gravelly soils, watering in dry weather, and mechanical injury on the collar render the plants to *Phomopsis* infection. Avoiding such cultural operations significantly reduced the incidence of collar canker. Surgical removal of affected tissue and rejuvenation pruning of unthrifty bushes helped to bring back the bushes to good health.^[5] Increased levels of potassium fertilizers improve the health and vigor of plants and develop tolerance to diseases like red rust and thorny stem blight.

Regulating shade by pollarding shade trees and annual lopping of side branches prior to monsoon reduce the incidence of blister blight. Apart from this, other cultural operations like adjusting the pruning time, black plucking, shorter plucking interval, and cutting of spraying lanes have an impact on blister blight control. The severity of blister blight is more in tea fields recovering from pruning due to the presence of abundant succulent, susceptible shoots. In dry weather pruning, the bushes recover at a time when weather conditions are not conducive for the development of blister blight. However, this resulted in a high incidence of branch and stem canker. Lime (slaked lime suspension) washing of the pruned bushes

minimizes the risk of sun-scorch injuries, besides controlling the growth of moss and lichen and inducing early bud break. Further, proper weed control regulates the microclimate and reduces the incidence of blister blight.

Chemical Control

Chemical control is the most effective and widely practiced disease management strategy in tea. Remarkable advances have been made to control root diseases by soil fumigation^[6] and soil drenching with systemic fungicides.^[7] The black root disease was effectively controlled by soil drenching with 0.3% mancozeb or 0.05% carbendazim. Protectant fungicides were not very effective in controlling stem diseases like wood rot, thorny stem blight, and stem canker, as the pathogens are deep seated. On the other hand, application of systemic fungicides arrested the growth of the pathogens and prevented further development of cankers caused by *M. theicola* and *Phomopsis*, wood rot, and thorny stem blight. However, the protection of the prune cut and other wounds by copper fungicides reduces the risk, as the stem pathogens mainly enter through wounds.

Copper fungicides were the most widely used fungicide to control various tea diseases. A combination of copper oxychloride and nickel chloride had been used for decades in tea plantations of southern India to control blister blight. The discovery of ergosterol biosynthesis inhibiting fungicides such as bitertanol, hexaconazole, propiconazole, and tridemorph opened a new era in the control of blister blight.^[8] These fungicides were effective even in very low concentrations. Combination of chlorothalonil and benomyl was effective in controlling anthracnose,^[9] benomyl, and thiophanate methyl against brown blight and white scab.^[1] Fungicides such as chlorothalonil, benomyl, and thiophanate methyl are widely used to control brown blight and anthracnose. Drenching of the bushes with copper fungicides (0.25% concentration) and also systemic fungicides like vitavax effectively controlled black rot disease. It was also found that drenching of bushes with mancozeb controlled grey blight disease; however, systemic fungicides like carbendazim and thiophanate methyl were superior to mancozeb.

Biological Control

Microbial interaction plays an important role in the natural control of plant diseases. However, in tea, biological control has been reported only in very few cases. *Trichoderma viride* and *Trichoderma harzianum* showed inhibitory activity against *Poria*

hypobrunea.^[10] Efficacy of biocontrol agents in controlling some of the primary and secondary root diseases, thorny stem blight, collar canker, and grey blight has been reported.^[7] Recently, Baby et al.^[11] identified efficient antagonistic strains of tea phylloplane bacteria and tested their biocontrol potential against blister blight.

CONCLUSIONS

Tea diseases are of great concern as they lead to capital loss, crop loss, and deterioration in the quality of tea. The perennial habit and monoculture habitat of tea plant provide a stable environment to many pathogens, enabling their easy dissemination. Most of the tea diseases are endemic in nature. Many diseases that were of minor importance earlier have become a major problem over the years. This is mainly because of the changes in weather factors and the cultural operations adopted in tea plantations. Introduction of continuous mechanized harvesting led to increased stress on plants, depletion in root carbohydrate reserves, and debilitation of plants, which in turn increased the incidence of many diseases. On the other hand, integration of mechanized harvesting with hand plucking prevented this ill effect on bush physiology. Fungicides have been extensively used to control tea diseases, but under the changing scenario of environmental protection it is essential to develop eco-friendly control strategies. Host plant resistance as well as cultural and biological control methods have the potential to reduce the incidence of many diseases. Research efforts are needed to develop an integrated system of disease management in tea by combining cultural and biological control methods with reduced use of fungicides.

REFERENCES

1. Chen, Z.M.; Chen, X.F. *The Diagnosis of Tea Disease and Their Control*; Shanghai Scientific and Technical Publishers: Shanghai, 1990.
2. Chandramouli, B. Common names of plant diseases. Diseases of tea (*Camellia sinensis* (L.) O. Kuntze). <http://www.apsnet.org/online/common/names/tea.asp> (accessed on August 2000).
3. Muraleedharan, N.; Chen, Z.M. Pests and diseases of tea and their management. *J. Plant. Crops* **1997**, *25*, 15–43.
4. Chandramouli, B. Root diseases of tea and their control. *J. Coffee Res.* **1988**, *18*, 1–57.
5. Venkata Ram, C.S. Rejuvenation of unthrifty tea. *Warta BPTK* **1976**, *2*, 133–140.
6. Shanmuganathan, N. Recent developments in the control of *Poria* root disease. *Tea Q.* **1964**, *35*, 22–30.
7. Baby, U.I. Diseases of tea. In *Advances in the Diseases of Plantation Crops and Spices*; Santhakumari, P., Ed.; International Book Distributing Co.: Lucknow, India, 2004; 321–344.
8. Baby, U.I.; Balasubramanian, S.; Ajay, D.; Premkumar, R. Effect of ergosterol biosynthesis inhibitors on blister blight disease, the tea plant and quality of made tea. *Crop Prot.* **2004**, *23*, 795–800.
9. Horikawa, T. Occurrence of tea grey blight and tea shoot blight caused by *Pestalotia longiseta* Spegazzini and control. Proceedings of the International Symposium on Recent Developments in Tea Production, Taiwan Tea Experimental Station: Taiwan 1988; 181–195.
10. Sathyanarayana, G.; Barua, G.C.S. Leaf and stem diseases of tea in NE India with reference to recent advances in control measures. *J. Plant. Crops* **1983**, *11*, 27–31.
11. Baby, U.I.; Balamurugan, A.; Jayaprakashvel, M.; Premkumar, R. Screening of phylloplane bacteria as biocontrol agents against blister blight disease of tea. *J. Plant. Crops* **2004**, *32* (suppl), 361–364.

Tea Insects: Ecology and Control

N. Muraleedharan

UPASI Tea Research Foundation, Valparai, Coimbatore,
Tamil Nadu, India

INTRODUCTION

The commercially cultivated tea plants are derived from *Camellia sinensis* (L.) O. Kuntze, the short-leaved “China” plants, *Camellia assamica* (Masters) Wight, the broad-leaved “Assam” plants, *C. assamica lasiocalyx* (Planchon ex Watt) Wight, the “Cambod” plants, and the numerous hybrids among them. According to a recent estimate, 1034 species of arthropods infest tea plants.^[1] Recently, a couple of reviews have been published on pests of tea in Asia and Africa.^[2,3] Among the insect pests, Lepidoptera is the largest order, containing 32% of the pest species, followed by Hemiptera with 27%. The dynamic adaptations of insects have enabled them to attack every part of the tea plant and the maximum number of pests occur on the foliage. Each geographic region has its own distinctive pest fauna, though several species have been recorded from more than one region. The number of insects and mites associated with tea plants depends on the length of time for which the crop is cultivated in that region. The area under tea became important only after allowing for “age effect” and latitude had no influence on species richness.^[4] In large tea-growing regions, saturation levels in the number of insect species is reached during a period of 100–150 years. The accumulation of arthropod species on tea is also influenced by plant age.

Crop loss in tea due to the ravages of pests is estimated to be 8% of the total loss.^[5] Another estimate showed a loss of 29 million kilograms of tea due to pests, accounting for 13% of the crop production in northeast India.^[6] Yet another assessment places this figure anywhere between 6% and 14%.^[7] Infestation by pests not only results in crop loss but also adversely affects the quality of processed tea.

PESTS OF TEA

Several species of phytophagous mites belonging to Tetranychidae, Tenuipalpidae, Eriophyidae and Tarsonemidae infest tea. The age of bushes in the pruning cycle influences the abundance of mites on tea plants. The population density of mites increases as the field advances in age from pruning. Population of mites is

less in shaded areas when compared to unshaded fields. The presence of shade trees tends to decrease leaf temperature by about 2°C below ambient temperature while in unshaded areas temperature of leaf may go up by 4°C.

Plant sap sucking mirids belonging to *Helopeltis*, commonly referred to as tea mosquitoes, are of great economic importance in most of Asia and Africa. The feeding activity of *Helopeltis* spp. is highest in the early mornings and late evenings. A single first-instar nymph of *Helopeltis theivora* can make as many as 80 feeding lesions in 24 hr. Scale insects and mealy bugs infesting leaves have a wide range of geographic distribution. Some of the important scale insects infesting tea are *Fiorinia theae*, *Pseudalacaspis duplex*, *Hemiberlesia cyanophylli*, and *Pulvinaria psidii*. The oriental tea tortrix *Homona magnanima* Diakonoff and *Adoxophyes* sp. cause extensive damage to tea in Japan and Taiwan while *Homona coffearia* Nietner is a serious pest in Sri Lanka. The flushworm, *Cydia leucostoma*, is the main tortricid pest in India, Bangladesh, and Indonesia. The gracillariid leaf roller *Caloptilia theivora* Walsingham, an economically important pest in Japan and Taiwan, is present in the other tea-growing countries of Asia also. The leaf folding caterpillars are more prevalent in fields recovering from pruning and construct several leaf cases. Many other lepidopterous pests belonging to Geometridae, Bombycidae, Lymantridae, Psychidae, and Limacodidae feed on the mature foliage of tea plants. Weevils belonging to *Mylloceris* feed on tea leaves in India, Sri Lanka, and China; *Systates smeii* attacks tea in Malawi and *Entypotrachelus meyeri* in Kenya. All these weevils eat young and old leaves and damage will be very severe if the attack is in newly planted areas or in nurseries. The scarabaeid beetles, *Mimela xanthorrhina* Hope, *Callistethus gemmula* (Arrow), and *Serica assamensis* Brenske feed on mature leaves, leaving the midrib and veins in such a manner that the damaged leaves present a skeletonized appearance.

Stems of the tea plants are affected by about 300 species of insects among which scale insects are the most dominant. They have been successful in invading not only leaves and stems but also roots. Several caterpillars eat the bark of bushes while a few others bore into the stems. *Indarbela theivora* Hampson is

the common bark-eating borer in India. In Malawi, *Teregra quadrangular* ringbarks young plants and on mature bushes the affected branches develop a callus around the wound and form a knot. The red coffee borer, *Zeuzera coffeae* Nietner, distributed throughout India, Sri Lanka, and Indonesia, bores into the stems and the tunnels may run through the main stem of young plants. In Japan, another species, *Zeuzera leuconotum*, causes similar damage. The oecophorid stem borer, *Casmara patrona* Meyrick, is seen in upper Assam as well as in China. The large hepialid stem borer, *Sahyadrassus malabaricus* (Moore), endemic to South India, is an occasional pest. In the temperate regions of China and Georgia, *Parametriotes theae* Kusnetsov damages tender stems. Certain scolytid beetles have become serious pests of tea. *Euwallacea fornicatus* (Eichhoff), the shot hole borer, is an important pest in Sri Lanka and South India and to a lesser extent in Indonesia. Certain species of termites cause direct injury by damaging the heartwood of stem while many others are scavengers, feeding on dead wood and bark. *Postelectrotermes militaris* (Desneux), *Neotermes greeni* Desneux, and *Glyptotermes dilatatus* Bugnion & Pop. are responsible for heavy crop loss in Sri Lanka. Large-scale removal of shade trees in that country made the environment more favorable for the live wood termites. The activities of these termites start in spring and continue into the cold weather. Scanty rainfall and prolonged dry conditions are favorable for their activity.

Insects damaging the roots of tea plants are very few. Grubs of *Holotrichia disparilis* are main pests of roots. Pseudococcids belonging to *Dysmicoccus* and *Crisicoccus* feed on roots, the former being more common on nursery plants. *Pseudococcus theacola* Green occurs on the roots of the mature plants in Darjeeling and Assam in India.

MANAGEMENT OF TEA PESTS

The literature on tea pest control is dominated by reports on chemical control. Broad-spectrum pesticides offer powerful incentives in the form of excellent pest control, increased yields, and reliable economic returns, but they have serious limitations. However, there are welcome efforts to adopt non-chemical control strategies and evolve an integrated pest management system. Attempts have been made to evolve an Integrated Pest Management (IPM) strategy for tea in Sri Lanka and India.^[8-10]

Use of resistant crop varieties is one of the acknowledged components of integrated pest management. Even low levels of resistance are important since the need for other control methods can be reduced. Different cultivars of tea with varying growth habits differ

in their susceptibility, resistance, or tolerance to pests. Erect leaves are preferred by sucking pests while semierect or horizontal broad-leaved cultivars are the choice of leaf rolling and chewing insects. Soft wooded tea plants are easily damaged by termites. Similarly, clones with high content of alpha spinasterol are susceptible to damage by the shot-hole borer, *Euwallacea fornicatus*. Being a perennial crop, research on clonal selection and breeding in tea is primarily aimed at the production of high-yielding and superior quality plants, with practically very little emphasis on resistance to pests.

Cultural practices in tea are governed by agronomic and economic considerations and they are unlikely to be changed solely because of the recommendations on pest control. Nevertheless, certain routine cultural operations such as plucking, pruning, shade regulation, and weed control can be manipulated to reduce the incidence of pests and the intensity of their damage. Manual removal of the larvae and pupae of many lepidopterous pests will go a long way in avoiding or reducing the number of applications of broad-spectrum insecticides. Populations of leaf folding caterpillars can be suppressed to a considerable extent by their manual removal during plucking. "Black plucking" in combination with insecticide application is effective for the control of *Helopeltis*. Harvesting of shoots at closer intervals will result in the removal of eggs of *Helopeltis* and also deny suitable material for feeding and oviposition.

The operation of pruning and the length of the pruning cycle are important factors in pest ecology. Pruning removes a large part of the foliage and stems along with the pests. However, the newly emerging foliage is nutritionally more attractive to certain insects such as aphids, thrips, flushworms, and leaf rollers. Introduction of an extended pruning cycle is supposed to have increased the activities of the tea mosquito in northeast India since the pest finds an undisturbed place for hibernation during winter and gets a supply of tender shoots earlier in the season. If the tea fields located near forests and prone to *Helopeltis* attack are kept under normal pruning, the activities of this pest can be reduced. Light pruning of the infested areas will reduce the intensity of attack in the following years as most of the eggs, embedded in the shoots, will be destroyed during pruning.

Minimizing new access points by removal of deadwood, cankers, and snags will lessen fresh attack by termites. Proximity of termite affected areas should be taken into consideration before selecting an area for new planting. Damage due to shot-hole borer is more severe in the third and fourth years of the pruning cycle, and there exists an exponential relation between the percentage of borer attack and the age of the field from pruning.

Application of higher levels of potassium fertilizers is known to reduce the incidence of pests and disease in several crops. Population density of the lesion nematode, *Pratylenchus loosi*, was significantly lower when potassium fertilizer was applied at higher doses. Application of a high rate of muriate of potash to the soil in the first year of the pruning cycle significantly reduced the infestation by shot-hole borer in India. Addition of castor oil cake and mahuva oil cake to the soil significantly suppressed the populations of *P. loosi* in Sri Lanka. Though this method alone may not be sufficient to achieve satisfactory control, soil amendment can be an important component of the control program against this nematode. Repeated applications of copper fungicides for the control of blister blight disease had resulted in the increased incidence of phytophagous mites on tea in Sri Lanka, India, and Indonesia. Copper oxychloride appears to enhance the fecundity of mites.

The introduction of the ichneumonid parasitoid, *Macrocentrus homonae* Nixon, from Java to Sri Lanka during 1935–1936 for the control of the tea tortrix, *H. coffearia*, is an excellent example of classical biological control in tea. Numerous biocontrol agents are active in tea fields, exerting a natural regulation of several pests.^[11] In fact, the minor status of several tea pests is due to the influence of an array of predators and parasitoids which are active in the tea ecosystem. Phytophagous mites infesting tea are preyed upon by several predatory mites, mostly belonging to Phytoseiidae and Stigmaeidae. Coccinellids are probably the second largest group of predators in tea fields. A complex of syrphid, coccinellid, and hemerobiid predators and aphidiid parasitoids exert significant influence on the populations of *Toxoptera aurantii*.

Field application of a mixture of granulosis viruses of smaller tea tortrix, *Adoxophyes orana* and *H. magnanima*, resulted in 50–80% control of both the tortricids in Japan. The bacterial insecticide *Bacillus thuringiensis* is effective against *H. magnanima*, *Caloptilia theivors*, and *Adoxophyes* sp. Its efficacy was also proved against *Andraca bipunctata* and *Buzura suppressaria*.

Components of the sex pheromones of the smaller tea tortrix *Adoxophyes* sp. have been identified and isolated. Extracts of virgin females and ultraviolet light traps were equally attractive to males of *A. orana* in the quantitative bioassay of the pheromones. Mass trapping with sex pheromones considerably reduced infestation by *Adoxophyes* both in Japan and in Taiwan.

A large number of insecticides have been recommended for pest control in tea. However, their use is governed by the maximum residue limit (MRL) of these insecticides on tea. Tea being an important export commodity, all precautions are taken by the tea-growing countries on the use of pesticides.

CONCLUSIONS

In most of the tea-growing countries, the tea plantations are in close proximity to the forest ecosystem and contribute greatly to the maintenance of terrestrial ecology by providing extensive land cover and by preventing soil erosion. The importance of pest control and the increasing awareness of the side effects of pesticides on the quality of the environment and safety to human health necessitate an “ecologically sound and economically feasible” approach to tea pest management. An in-depth understanding of the influence of natural enemies, including the entomopathogens of tea pests, is essential if biocontrol strategies are to be successfully implemented. Several of the agronomic practices can be manipulated to reduce the population density of pests without compromising on yield and quality of tea. Semiochemicals can be a significant, complementary tool in the management of pests, but we need to strengthen our research efforts on this subject. Pesticides will continue to play a significant role in tea pest management and therefore there is an urgent need to generate more data on the pattern of their degradation on tea plants to establish their MRLs on processed tea.

REFERENCES

1. Chen, Z.M.; Chen, X.F. An analysis of world tea pest fauna. *J. Tea Sci.* **1989**, *9* (1), 73–88.
2. Muraleedharan, N. Pest control in Asia. In *Tea—Cultivation to Consumption*; Willson, K.C., Clifford, M.N., Eds.; Chapman & Hall: London, 1992; 375–412.
3. Rattan, P.S. Pest and disease control in Africa. In *Tea—Cultivation to Consumption*; Willson, K.C., Clifford, M.N., Eds.; Chapman & Hall: London, 1992; 331–374.
4. Banerjee, B. An analysis of the effect of latitude, age and area on the number of arthropod pest species of tea. *J. Appl. Ecol.* **1982**, *18*, 339–342.
5. Cramer, H.H. *Plant Protection and World Crop Production*; Bayer: Leverkusen, Germany, 1967.
6. Glover, P.M.; Das, G.M.; Mukherjee, T.D. Pesticide residues and taint in tea. *SPAN* **1961**, *4* (3), 137–140.
7. Banerjee, B. Pesticides and pesticide residues in tea. *Two Bud* **1976**, *23* (2), 342–345.
8. Calnaido, D. An ecological approach to pest management in tea. *Tea Q.* **1971**, *42*, 175–181.
9. Sivapalan, P.; Deluchhi, V. Integrated approach in tea pest control in Sri Lanka. *Tea Q.* **1975**, *45*, 80–90.
10. Muraleedharan, N.; Selvasundaram, R. An IPM package for tea in India. *Planters' Chron.* **2002**, *98* (4), 104–124.
11. Muraleedharan, N.; Selvasundaram, R.; Radhakrishnan, B. Parasitoids and predators of tea pests in India. *J. Plant. Crops* **2001**, *29*, 1–10.

Temperate-Climate Fruit Crop Pest Management: Plant Pathogens

David F. Ritchie

*Department of Plant Pathology, North Carolina State University,
Raleigh, North Carolina, U.S.A.*

INTRODUCTION

Temperate-climate fruit crops, such as pome fruits (e.g., apples and pears), stone fruits (e.g., peaches, nectarines, cherries, plums), brambles (e.g., blackberries, raspberries), blueberries, grapes, kiwifruit, and strawberries, generally are among the highest value crops grown (\$1000 per hectare). However, establishment costs are also high, and except for strawberries, most fruit crops do not begin to bear until at least two years after establishment; consequently, growers need to do as much as possible to protect their investment. It is therefore important, firstly, to select a planting site with good air movement and with access to a full day of sunshine to reduce risks from late spring freezes and to promote optimal coloring and sugar content of fruit as ripening occurs. Secondly, the soil type and structure are also important for good plant growth and the reduction of soil-borne diseases. Thirdly, a certified pathogen-free plant material should be used.

Quality, including “eye appeal,” is essential for fruits destined for fresh market. Pathogens, especially foliar pathogens and/or some viruses, can affect marketable yield, thus decreasing profits. Thus, for fruit to be competitive, disease-causing pathogens and other pests must be managed successfully. Disease management is compounded by the array of pathogens that can attack the fruit, foliage, branches, and roots. Occurrence and severity of these diseases are greatly influenced by moisture and temperature. They are primarily managed by a combination of cultural and chemical controls.

PATHOGENS

Representatives of all major pathogen groups, including fungi, bacteria, nematodes, viruses, and phytoplasmas (formally known as mycoplasma-like), cause diseases of fruit crops. Fruit diseases result in direct crop loss; however, viruses, phytoplasmas, other foliar and soil-borne diseases, and nematodes indirectly affect fruit yield and quality as well as the productive longevity of the plant. Many of the pathogens that

infect fruit also infect leaves, branches, and stems; thus, the pathogens are able to complete their life cycles on the crop or on nearby reservoir and alternate hosts. Nematodes are parasites that may directly damage the plant by causing severe root damage (e.g., root knot), function as predisposition agents (e.g., lesion and ring nematodes) of the plant to other biological and environmental factors, and can serve as virus vectors (e.g., dagger nematodes). Some of the most common fruit diseases and their pathogens/parasites are listed in Table 1.

MANAGEMENT STRATEGIES

Successful disease management starts with the selection of varieties adapted to the geographical growing region. Equally important is the selection of an appropriate site for growing the fruit crop. Fruit crops and varieties planted in areas and soils to which they are not adapted do not grow well and are more prone to diseases. Once the varieties and a growing site are selected, disease management is built on four basic principles: the use of genetic resistant plants, if available, adapted to the region; the use of disease/pathogen-free plants or planting material, cultural and chemical control of pathogens, and vectors of pathogens; and the use of good sanitation practices throughout the year.

Biological control agents (BCAs) have been most successful where traditional chemical controls are lacking or cannot be used because of concerns for human health and safety reasons. One of the most successful uses of a BCA has been in the management of the bacterial disease crown gall. BCAs also have shown efficacy for management of fire blight, some foliar and fruit fungal diseases, and postharvest fruit diseases.

Soil-borne problems caused by nematodes and fungi pose management challenges with the loss of soil fumigants because of environmental and human safety concerns. Thus, management tactics focus on planting site selection for pathogen avoidance when possible; the use of soil amendments, groundcover, and secondary host management; host resistance; and biocontrol.

Table 1 Common diseases and pathogens of fruit crops and the major plant organ(s) infected^a

| Crop and disease | Pathogen or parasite | Primary plant organ(s) infected | | | | |
|---|--|---------------------------------|-------|--------|----------------------------------|--------------------|
| | | Blossom | Fruit | Leaves | Stems, branches, limbs, or trunk | Crown and/or roots |
| <i>Brambles</i> | | | | | | |
| Anthracnose | <i>Elsinoe veneta</i> | + | + | + | + | |
| Crown/cane gall, hairy root | <i>Agrobacterium</i> spp. | | | | + | + |
| Cane blight and canker | <i>Botryosphaeria</i> sp. <i>Botrytis</i> sp. <i>Leptosphaeria</i> sp. | | | | + | |
| Rusts | <i>Arthuriomyces</i> sp. <i>Phragmidium</i> sp. <i>Pucciniastrum</i> sp. | | | + | + | |
| Fruit rots | <i>Botrytis</i> sp. <i>Rhizopus</i> spp. | | + | | | |
| Root rot | <i>Phytophthora</i> spp. | | | | | + |
| Root-lesion nematode | (<i>Pratylenchus</i> spp.) | | | | | + |
| Dagger nematode | (<i>Xiphinema</i> spp.) | | | | | + |
| Virus and phytoplasma diseases ^b | | + | + | + | + | + |
| <i>Blueberry</i> | | | | | | |
| Mummy berry | <i>Monilinia</i> sp. | + | + | + | + | |
| Fruit rot | <i>Phomopsis</i> spp. <i>Botrytis</i> spp. <i>Colletotrichum</i> spp. | | + | | | |
| Stem cankers and leaf spots | <i>Botryosphaeria</i> spp. <i>Phomopsis</i> spp. <i>Septoria</i> spp. | | | + | + | |
| Root rot | <i>Phytophthora</i> spp. | | | | | + |
| Virus and phytoplasma diseases ^b | | + | + | + | + | |
| <i>Grapes</i> | | | | | | |
| Powdery mildew | <i>Uncinula necator</i> | + | + | + | + | |
| Downy mildew | <i>Plasmopara viticola</i> | + | + | + | + | |
| Fruit rot | <i>Botrytis</i> sp. <i>Guignardia</i> sp. <i>Colletotrichum</i> spp. | | + | + | | |
| Crown gall | <i>Agrobacterium</i> sp. | | | | + | + |
| Pierce's disease | <i>Xylella fastidiosa</i> . | | | + | + | |
| Root-knot nematode | <i>Meloidogyne</i> spp. | | | | | + |
| Dagger nematode | <i>Xiphinema</i> spp. | | | | | + |
| Virus and viruslike diseases ^b | | | + | + | + | + |
| <i>Kiwifruit</i> | | | | | | |
| Fruit rot | <i>Botrytis cinerea</i> | + | + | | | |
| Bacterial blight and bleeding canker | <i>Pseudomonas syringae</i> <i>Pseudomonas viridiflava</i> | | | | + | |
| Crown gall | <i>Agrobacterium</i> sp. | | | | + | + |
| Crown and root rot | <i>Phytophthora</i> spp. <i>Armillaria</i> sp. | | | | | + |
| <i>Pome fruits</i> | | | | | | |
| Scab | <i>Venturia</i> spp. | | + | + | + | |
| Rusts | <i>Gymnosporangium</i> spp. | | | + | + | |
| Powdery mildew | <i>Podosphaera</i> spp. | | + | + | + | |
| Fire blight | <i>Erwinia amylovora</i> | + | + | + | + | |

(Continued)

Table 1 Common diseases and pathogens of fruit crops and the major plant organ(s) infected^a (Continued)

| Crop and disease | Pathogen or parasite | Primary plant organ(s) infected | | | | |
|---|-----------------------------------|---------------------------------|-------|--------|----------------------------------|--------------------|
| | | Blossom | Fruit | Leaves | Stems, branches, limbs, or trunk | Crown and/or roots |
| Fruit rots and cankers | <i>Botryosphaeria</i> spp. | | + | + | + | |
| | <i>Colletotrichum</i> spp. | | + | + | + | |
| Crown and root rot | <i>Phytophthora</i> spp. | | | | | + |
| | <i>Armillaria</i> spp. | | | | | + |
| Root-lesion nematode | <i>Pratylenchus</i> spp. | | | | | + |
| Dagger nematode | <i>Xiphinema</i> spp. | | | | | + |
| Virus and phytoplasma diseases ^b | | | + | + | + | + |
| <i>Strawberry</i> | | | | | | |
| Fruit rot | <i>Botrytis</i> sp. | + | + | | | |
| | <i>Colletotrichum</i> spp. | | + | + | | |
| | <i>Phytophthora</i> sp. | | + | | | |
| | <i>Rhizopus</i> sp. | | + | | | |
| | <i>Phomopsis</i> sp. | + | + | + | | |
| Powdery mildew | <i>Sphaerotheca</i> sp. | | | + | | |
| Red stele | <i>Phytophthora</i> sp. | | | | | + |
| Angular leaf spot | <i>Xanthomonas</i> sp. | | + | + | + | |
| Leaf, stem nematode | <i>Ditylenchus</i> sp. | | | + | + | |
| Root-knot nematode | <i>Meloidogyne</i> spp. | | | | | + |
| Root-lesion nematode | <i>Pratylenchus</i> spp. | | | | | + |
| Dagger nematode | <i>Xiphinema</i> spp. | | | | | + |
| Virus and phytoplasma diseases ^b | | + | + | + | + | |
| <i>Stone fruits</i> | | | | | | |
| Brown rot | <i>Monilinia</i> spp. | + | + | + | + | |
| Leaf curl | <i>Taphrina</i> sp. | | + | + | | |
| Powdery mildew | <i>Podosphaera</i> spp. | | + | + | | |
| | <i>Sphaerotheca</i> spp. | | + | + | | |
| Bacterial canker | <i>Pseudomonas syringae</i> | + | + | + | | |
| Bacterial spot | <i>Xanthomonas arboricola</i> pv. | | + | + | + | |
| Crown gall | <i>Agrobacterium</i> sp. | | | | + | + |
| Root rot | <i>Armillaria</i> spp. | | | | | + |
| | <i>Phytophthora</i> spp. | | | | + | + |
| Root-knot nematodes | <i>Meloidogyne</i> spp. | | | | | + |
| Ring nematode | <i>Criconemella</i> sp. | | | | | + |
| Lesion nematode | <i>Pratylenchus</i> sp. | | | | | + |
| Dagger nematode | <i>Xiphinema</i> sp. | | | | | + |
| Virus and phytoplasma diseases ^b | | | + | + | + | + |

^aListing of diseases and pathogens is not meant to be exhaustive. More information may be obtained in other sources such as the Crop Diseases Compendium series published by the American Phytopathological Society, St. Paul, MN, USA.

^bFruit crops, most of which are vegetatively propagated, are prone to numerous viral, viral-like, and phytoplasma diseases.

Although the use of fruit varieties having disease resistance is ideal, it is a difficult goal to achieve because of the characteristics of both the crop and the pathogens. Most fruit crops do not come into bearing for several years; they do so when a dormancy period is fulfilled. Thus, breeding programs for fruit crops are long-term endeavors. Additionally, many varieties are selected for “consumer appeal” and shipping-and-storage qualities rather than disease resistance. For complete reliance upon host resistance, normally a high level of resistance is needed because of market

demands for blemish-free fruit. Also, fruit crops are affected by multiple pathogens; thus, resistance to one pathogen still may not negate the need to use fungicides to manage others. On the pathogen side, pathogen populations evolve or are selected; they can defeat host resistance. High levels of pathogen resistance often are associated with genotype-specific resistance (i.e., vertical resistance), which is conferred by single genes and is prone to non-durability. Biotechnology and the use of transgenic methods hold great promise for developing disease resistance in crops that

have long cycles of development when traditional breeding methods are used. However, there may be potential biological (e.g., lack of single-gene durability) and social (e.g., lack of consumer acceptance) pitfalls regarding the use of transgenics.

Starting with certified disease-free plants is essential for successful production of fruit crops. This is essential for management of virus-caused diseases. Once the crop is established, sanitation plays a major role in successful disease management. Most fruit crops are perennial, and many pathogens survive from one bearing season to the next either on the host plant or within or near the crop site in reservoir and alternate hosts. Thus, sanitation practices that remove diseased fruit and plant parts from the crop area aid in inoculum and usually subsequent disease reduction.

Fungicides

Fungicides have traditionally and still play an important role in disease management of fruit crops because many diseases cannot be controlled adequately otherwise. The appearance of disease symptoms and signs shows that the pathogen has successfully infected the host; thus, monitoring for disease symptoms and signs per se is not adequate for managing many diseases. This is particularly true for diseases of fruit that develop rapidly, causing rots such as brown rot of stone fruits. In contrast, some diseases, such as powdery mildew, occur on the foliage or do so prior to infecting fruit. Other diseases have latent periods of days or weeks between infection and the occurrence of symptoms, thus allowing time for use of an eradicant fungicide before economic damage occurs. Development of fungicides having eradicated or curative properties in the last half of the 20th century further stimulated the development of forecast models. Although models may accurately predict the occurrence of disease, they have little practical value if effective interventions (e.g., eradicant fungicides) are not available to either prevent or eradicate the infection.

Forecast Models

Most disease-forecast models are based upon defining the relationship between particular weather conditions as they influence the infection process and crop phenology. The Mill's model, published in 1944, was designed to aid in timing sulfur dust applications for the control of apple scab. It was based on the concept that the occurrence and severity of apple scab was related to the time length of leaf wetness and the temperature during the wetting period. Based on the concept of this model, predictive disease models have been

developed for many other diseases of fruit crops. These include fire blight of pome fruits, rusts and many of the fruit rot and blemishing diseases of apples, downy and powdery mildews and fruit rots of grapes, and leaf spots of cherries. These models have been used with varying levels of disease management and economic success. Some of the success or failure of predictive models is associated with the number of diseases occurring on a given fruit crop, the accuracy in measurement of environmental conditions and weather forecasts, the severity of conditions for infection and disease development, and the effectiveness of intervention tactics.

INFORMATION DELIVERY

Since the mid-1990s, the World Wide Web has established itself as a rapid source of information for aiding in making disease management decisions. Information, such as aid in diagnosis, review of management strategies, the latest on pesticides, access to scientific and production journals, and real-time weather forecasts, are available. Some of this information can be available at no cost, while others are available on a subscription basis.

CONCLUSION

The management of fruit crop diseases will become more complex, but will increase in efficiency. Improved methods for pathogen detection and quantification combined with more accurate weather forecasts will aid in the prediction of infection and subsequent disease occurrences. Crop protection chemicals that are highly specific and have low toxicity to non-target organisms and the environment will continue to be developed and are incorporated into spray programs. Synthetic chemicals and BCA that activate natural plant defense systems will receive increased investigation and applications. The use of molecular biology will aid in shortening the time required to breed disease-resistant varieties and possibly in the development of highly effective BCAs.

BIBLIOGRAPHY

- Briggs, A.R.; Grove, G.G. Role of the World Wide Web in extension plant pathology: case studies in tree fruits and grapes. *Plant Dis.* **1998**, *82*, 452–464.
- Funt, R.C.; Ellis, M.A.; Madden, L.V. Economic analysis of protective and disease-forecast-based fungicide spray programs for control of apple scab and grape black rot in Ohio. *Plant Dis.* **1990**, *74*, 638–642.
- Janisiewicz, W.J. Biocontrol of postharvest diseases of temperate fruits—challenges and opportunities. In *Plant-Microbe*

- Interactions and Biological Control*; Boland, G.J., Kuykendall, D., Eds.; Marcel Dekker: New York, 1998; 171–198.
- Jones, A.L., Aldwinckle, H.S., Eds.; *Compendium of Apple and Pear Diseases*; APS Press: St. Paul, MN, 1990; 100.
- Ogawa, J.M., Zehr, E.I., Bird, G.W., Ritchie, D.F., Uriu, K., Uyemoto, J.K., Eds.; *Compendium of Stone Fruit Diseases*; APS Press: St. Paul, MN, 1995; 98 pp.
- Sikora, R.A. Management of the antagonistic ecosystem for the biological control of plant parasitic nematodes. *Annu. Rev. Phytopathol.* **1992**, *30*, 245–270.
- Steiner, P.W.; Lightner, G. *MARYBLYT: A Predictive Program for Forecasting Fire Blight Disease in Apples and Pears. Version 4.0 of a Copyrighted Computer Program*; University Maryland: College Park, 1992.
- Stockwell, V.O.; Johnson, K.B.; Loper, J.E. Establishment of bacterial antagonists of *Erwinia amylovora* on pear and apple blossoms as influenced by inoculum preparation. *Phytopathology* **1998**, *88*, 506–513.
- Sutton, T.B. Changing options for the control of deciduous fruit tree diseases. *Annu. Rev. Phytopathol.* **1996**, *34*, 527–547.
- UCPMG. Pest management and identification. Pests of agricultural crops, floriculture, and commercial turf. In *UC Pest Management Guidelines*; 1999. Davis, CA <http://www.ipm.ucdavis.edu/PMG/crops-agriculture.html> (April 5, 2002).

Tillage and Cultivation

Joseph Ingerson-Mahar

Rutgers University, New Brunswick, New Jersey, U.S.A.

INTRODUCTION

All crop management decisions begin with the soil or crop substrate. How the soil medium is managed has a profound effect on crop production and pest management. In many instances the role of tillage in modern-day agriculture is more important for reducing crop production costs and limiting erosion than it is in managing pests. Regional and global marketing of produce demands that commercial farmers are efficient managers of their crops while maintaining good stewardship of their cropland to keep it as productive as possible. For these reasons more and more emphasis has been placed on modifying tillage practices in order to reduce production costs and maintain soil tilth and fertility while providing quality produce. Pest-management practices must be modified according to the tillage system used and the subsequent pest population that develops.

TILLAGE SYSTEMS

Two primary tillage systems with variations are currently being utilized in modern agriculture: conventional tillage and conservation tillage. Conventional tillage, i.e., using a plow to turn over the soil and then using other tillage devices to break up clods of soil, is an ancient practice and was the primary tillage method until about 30 to 40 years ago. Tilling the soil was found to improve seed germination by providing a better seed bed.^[1] Now in addition to agronomic benefits conventional tillage is seen as providing pest-management benefits of certain perennial weeds, plant diseases, and insects. Tillage disrupts long-term cycles and favors small organisms with short life cycles and rapid dispersal.^[2] Disruption of life cycles occurs both by burying and by exposing pests to harsh weather conditions and predators. One way to help prevent disease carryover to succeeding crops is to bury or plow under disease-infected crop residues.^[3,4] Plowing or disking allows birds to provide a pest-management service in picking up exposed soil insects such as caterpillars, wireworms, and white grubs.^[5] One of the more memorable scenes of American agriculture is a field being tilled in the spring with a flock of birds following behind the tractor and disk (Fig. 1). Unfortunately,

plowing and disking the soil has nearly always been a high-energy and time-consuming practice that frequently causes as many problems (such as compaction layers, soil crusting, and wind and water erosion) as it solves.^[1]

Conservation tillage, restricted tillage including no-till and minimum till practices (no plowing or disking), reduces the amount of field traffic, soil erosion, and soil compaction but tends to rely more heavily on pesticides, especially herbicides, for pest control. The conversion to conservation tillage began when it was found to be less expensive and detrimental to soil structure than conventional tillage. Adoption of conservation tillage appears to be inversely proportional to the size of the farm: the larger the farm the more likely conservation tillage is practiced.^[6] The Natural Resources and Conservation Service, Department of Agriculture, has developed recommendations for ideal amounts of residue cover for different soils in conservation tillage that will help control soil erosion.^[7] It should be emphasized that the long-term benefits of reduced tillage outweigh the potential for increased pesticide use, but there are inherent pest risks with this tillage system. Conservation tillage favors the build-up of long-term soil pests and creates suitable environments for residue inhabiting pests including species of economically damaging caterpillars and stinkbugs, slugs, mice, woody perennials such as briars and dewberry, and diseases.^[8] Conversely, various studies have illustrated that some foliar and root-infesting insect pests (European corn borer, southern corn stalk borer, and western corn rootworm, as examples) become less important in conservation tillage fields. Another benefit of conservation tillage is that populations of large seeded weed species, such as jimson weed, velvet leaf, morningglory, and giant ragweed, tend to decline.^[9]

It is essential that farmers understand the risk of pest pressure in conservation tillage, especially with respect to insect pests. Where wireworms and white grubs are already present the switch to minimum-tillage practices enhances their populations. Similarly in no-till situations the new crop is directly seeded into the weed and old crop residue. An herbicide is then applied which kills the weeds before the crop seeds have emerged. While this is an efficient use of time and energy it also exposes the newly emerging crop to insect pest pressure: wireworms and white grubs



Fig. 1. Photograph courtesy of Don Prostack.

may feed on the roots and below ground portion of the plants, cutworms and stalk borers harbored in grassy clumps turn to the new crop as their weedy food supply dies, armyworms find grass crops (corn and sorghum/sudangrass) attractive in this situation.^[5] In the Midwest and Northeast where no-till corn is grown, immature stages of *Euschistis* stink bugs will also switch feeding from dying broadleaf weeds to late planted corn, sometimes resulting in severe crop loss.

SPECIAL PEST PROBLEMS

Fields infested with plant parasitic nematodes present a special problem. While they inhabit the soil similar to soil insects, rather than suppressing nematode populations tillage operations of any sort may actually increase the area of infestation by spreading the nematodes through the soil and then to other fields on the tillage equipment. Patterns of nematode infestations often follow the same direction as the path of field equipment across the field. Sanitation of tillage equipment is paramount for preventing the spread of nematodes between fields.

A similar argument can be made for perennial weeds that are capable of reproducing via rhizomes, e.g., Johnsongrass, quackgrass, and nutsedge. As portions of the rhizomes are broken off by tillage equipment they are easily spread through the field where they will take root.

Soil-borne fungi such as *Fusarium* and *Verticillium*, which cause wilt and death of plants, may persist in the soil for years as long as suitable hosts exist, either crops or weeds. It is unclear whether tillage practices can alter the soil environment to inhibit the persistence of these disease-causal organisms. However, as with nematodes, tillage equipment will help move these pathogens within and between fields.^[4]

CULTIVATION

Cultivation of crops using a tractor-mounted device with tines or prongs set to the width of the crop rows usually is done to loosen the soil and to uproot weeds, which augments herbicide use. Farmers often cultivate for weed control just before the crop becomes too large to move equipment through it relying upon the size and density of the crop plants to shade out remaining weeds in the field. Additional benefits include disruption of soil insects and further oxidation of organic matter which releases plant nutrients into the soil. All of these actions benefit the crop. However, excessive or deep cultivation may be detrimental to the crop by severing roots and providing entry points for disease organisms.^[4]

Summer following a field is tilling an empty field for the purpose of reducing weed infestations, disrupting soil insects, or even the eggs of grasshoppers.^[5] For weed management the field is worked at times through the growing season to prevent weeds going to seed and to kill seedlings as they emerge. Both objectives serve to decrease the seed bank in the field. Timing is critical in disrupting insect life cycles.^[5] In order to destroy grasshopper eggs cultivation must be done prior to the eggs hatching. Wireworms prefer moist soil and are sensitive to dehydration. Cultivating a fallow field where the upper soil layer lacks moisture will cause little wireworm mortality.

CONCLUSION

As important as it is for farmers to prevent erosion and preserve their soils they must also consider the management of their crops and know the benefits and risks of the particular tillage system they use. Not only should fields be periodically tested for nutrient levels and pH but also for nematode populations. Should

economically damaging levels of plant parasitic nematodes exist then the appropriateness of tillage practices will have to be reviewed for the infested fields. Likewise, if particular fields are subject to frequent soil insect infestations, plant diseases that overwinter on crop residue, or harbor herbicide-resistant weeds then conservation tillage may not be the most appropriate tillage method. As with any pest-management technique, the initial step in managing a pest depends upon the accurate identification of the pest and being familiar with the biology of the pest so that use of the appropriate tillage technique can be made.^[5]

Regardless of which tillage system is used it is one component of a complex series of management decisions designed to improve or maintain crop yield and quality. Integrating the appropriate tillage practice with crop rotation, variety selection, and other considerations becomes a powerful mechanism for reducing the economic impact of crop pests. Farmers must use the tillage system that best fits their crops, soil conditions, and pest regimes.

REFERENCES

1. Magdoff, F.; van Es, H. *Better Soils for Better Crops*, 2nd Ed.; SARE Series; Sustainable Agriculture Network: Burlington, VT, 1995; Vol. 4.
2. Doran, J.W.; Werner, M.R. Management and soil biology. In *Sustainable Agriculture in Temperate Zones*; Francis, C.A., Flora, C.B., King, L.D., Eds.; John Wiley and Sons, Inc.: New York, 1990; 205–230.
3. <http://www.pestmanagement.rutgers.edu/IPM/Vegetable/guidelines.htm> (accessed October 2002).
4. Agrios, G. *Plant Pathology*, 2nd Ed.; Academic Press: New York, 1978.
5. Sub-Committee on Insect Pests Cultural Control. In *Insect-Pest Management and Control*; Principles of Plant and Animal Pest Control Series; Publication 1695; National Academy of Sciences: Washington, DC, 1969; Vol. 3, 208–242.
6. Edwards, C.R.; Ford, R.E. Integrated pest management in the corn/soybean agroecosystem. In *Food, Crop Pests, and the Environment*; Zalom, F.G., Fry, W., Eds.; APS Press: St. Paul, MN, 1992; 13–55.
7. Impacts of Residue Management Practices. In *Core 4: Conservation Practices, Common Sense Approach to Natural Resource Conservation*; USDA: Washington, DC, 1999; 3–47.
8. Croft, B.A.; MacKenzie, D.R.; Cook, R.J. Biological constraints. In *Crop Productivity—Research Imperatives Revisited, an International Conference Held at Boyne Highlands Inn, October 13–18, 1985, and Airlie House, December 11–13*; Gibbs, M., Carlson, C., Eds.; 1985; 177–195.
9. Buhler, D.D. Effects of tillage and light environment on emergence of 13 annual weeds. *Weed technology*. *J. Weed Sci. Soc. Am.* **1997**, *11* (3), 496–501.

Unisexual Parasitoids in Biological Control

Richard Stouthamer

Department of Entomology, University of California, Riverside, California, U.S.A.

INTRODUCTION

Normally, parasitoid wasps produce sons from unfertilized eggs and daughters from fertilized eggs. This reproduction is a mix of parthenogenetic (i.e., production of offspring from unfertilized eggs) and sexual reproduction (i.e., the production of offspring from fertilized eggs). However, there are also parasitoids that produce only daughters and for their production no fertilization is needed; this is called unisexual reproduction.

Unisexuality is considered to be a desirable trait in parasitoids used for biological control. Compared to sexual parasitoids, unisexual forms are thought to: 1) have a higher rate of population increase because all the offspring are females; 2) be cheaper to produce in mass rearing, because none of the expensive hosts are wasted for the production of males; 3) be better colonizers because they never need to find mates.^[1] Many parasitoid species have forms or populations that can reproduce unisexually. Unisexual reproduction is known from at least 158 parasitoid wasp species.^[1] The frequency of unisexuality appears to be particularly high, at least 15%, in species applied in biological control.^[2]

Unisexuality was studied by many of the pioneers in biological control. They found that unisexual species that normally only produce daughters start producing males if their mothers were exposed to elevated rearing temperatures.^[3] Later, this finding was used to show that in many parasitoids the unisexual reproduction is caused by a bacterial infection.^[4] Two different bacterial groups are known to cause unisexuality: *Wolbachia*^[5] and *Cytophaga*-like bacteria.^[6] In some cases these infections can be transmitted to sexual parasitoids making them unisexual.^[7,8] This has opened the interesting possibility that in the near future it may be possible to render parasitoids unisexual for use in biological control.

ADVANTAGE OF UNISEXUAL REPRODUCTION FOR BIOLOGICAL CONTROL: FACT OR FICTION?

In theorizing about the advantages of unisexual reproduction over sexual reproduction, the assumptions are

made that the only difference between a sexual and a unisexual form is the fact that the unisexual form produces only daughters, whereas the sexual form uses some of her eggs to produce males. The biological control pioneers Timberlake and Clausen in 1924 used these assumptions to predict the population growth of the unisexual vs. sexual forms of a species.^[9] They calculate that at the end of one season the offspring of a single unisexual female of the parasitoid *Aschryso-pophagus modestus* would consist of 312,500,000 individuals, whereas the offspring of a single sexual female, producing offspring with a sex ratio of 75% females, would be only 74,200,000 individuals. Are these assumptions warranted? In general, there are not many cases where we can test these assumptions, but in several species of *Trichogramma* the offspring production of sexual and unisexual forms has been compared. In the laboratory the unisexual forms produce less offspring than the sexual forms. In some cases the unisexual forms produced even fewer daughters than the sexual females did. However, this may be specific for the *Trichogramma* case where the *Wolbachia*-induced unisexuality is unusual. In small-scale greenhouse experiments, sexual and unisexual forms of *Trichogramma* were tested against each other to determine whether the unisexual form would have a higher population growth rate.^[10] This experiment was done by placing cards with patches of host eggs on the tomato plants and releasing known numbers of sexual and unisexual females in the greenhouse. The results of these tests showed that the sexual and unisexual females were equally capable of finding the egg cards, but that the sexual forms laid more eggs per host patch. The results show that the growth rate of these wasps under these biocontrol conditions depends on the distribution of the hosts on the plants. If the hosts occur as single eggs then the unisexual form will have a higher rate of increase, whereas if the hosts are clustered in patches the sexual form will have a higher rate of increase. In other species the infection with *Wolbachia* does not appear to have such a negative impact on the wasp's offspring production, and it is therefore assumed that in those cases the unisexual form will have a much higher rate of population growth than the sexual form.^[5]

The assumption is also made that the cost of producing a single unisexual female is less than that of a

sexual female. In the case of unisexual wasps all hosts will be used for producing females, whereas sexual wasps use some of the hosts to produce males. However, if the unisexual form has a higher rate of mortality during the preadult stage this may influence the relative cost of producing a female. In general, it can be shown that as long as the frequency of larval mortality of female unisexual wasps is less than the fraction of males in the sexual population, the cost of producing a unisexual female will be lower.^[1] Although the superiority of unisexuals as colonizers has never been experimentally verified, it is very likely to be a clear advantage for the unisexual forms. Low densities of wasps during population establishment may make it difficult for sexual wasps to encounter a mate; unisexual wasps can produce offspring as long as they encounter hosts. These mate-finding problems may be important in biological control efforts.

Similar problems during the establishment of laboratory cultures may explain the relatively high frequency of unisexuality in wasps used in biological control.^[2] It is difficult to establish a sexual culture if only a few individuals are collected. At least one male and one female of the sexual forms are needed simultaneously to establish a sexual culture, whereas a single unisexual female can establish a population. Very small numbers of unisexual wasps released can lead to establishment and spread of the parasitoids. In Canada after the release of only two females of the unisexual species *Apanteles pedias*, the parasitoid population spread rapidly over a large area (see Ref.^[1]).

Are unisexual species more successful in biological control? While there appear to be many advantages for the use of unisexual species in biocontrol, there are no rigorous experiments that have tested this thesis. In only a few cases have sexual and unisexual forms of the same species been available for biocontrol. Experiments have never been specifically conducted to test the performance of the different reproductive modes, with the exception of the *Trichogramma* experiments described above.^[10] However, the few cases where some sort of comparison was possible do not show a clear advantage for the unisexual forms (for review see Ref.^[1]).

CAUSES OF UNISEXUALITY

Unisexuality in wasps is a trait that is in some species inherited through the genes of the wasps, but in the majority of cases unisexuality is caused by infection with symbiotic bacteria.^[5] Infections with symbiotic bacteria are common in insects. Two bacterial groups are symbionts of many different insects: *Wolbachia*^[5] and *Cytophaga*-like bacteria.^[6] These bacteria are inherited from mother to offspring through the

cytoplasm of the eggs. They have many different effects on their host, but here we will only discuss the induction of unisexuality. *Wolbachia* that induce unisexuality do so by manipulating the chromosome behavior in the first mitotic division of the egg. They cause an abortion of the first mitotic anaphase, which allows the two sets of the chromosomes to remain in the same nucleus and, consequently, these eggs develop as females.^[5]

The bacteria causing unisexuality can be transmitted from infected wasps to uninfected wasps either through microinjection^[7] or through super parasitization.^[8] In some cases they will also induce unisexuality in their new hosts. Although this transmission from one species to another appears feasible in the laboratory, in the field such transmissions are most likely very rare and may often have a negative impact on the new host. With the further development of these microinjection techniques it may be possible to make species unisexual that are considered for application in biological control.

CONCLUSION

Unisexual reproduction has in theory many advantages for parasitoids used for biological control. In the near future we will be able to render potential biological control agents unisexual. Our ability to render wasps unisexual will allow us to do the required experiments to show that the assumed advantages of unisexual reproduction indeed translate into better biological control.

REFERENCES

1. Stouthamer, R. The use of unisexual wasps in biological control. In *Quality Control and Production of Biological Control Agents. Theory and Testing Procedures*; van Lenteren, J.C., Ed.; CABI: Wallingford, UK, 2003; 93–113.
2. Luck, R.F.; Stouthamer, R.; Nunney, L. Sex Determination and sex ratio selection in parasitic hymenoptera. In *Evolution and Diversity of Sex Ratio in Haplodiploid Insects and Mites*; Wrensch, D.L., Ebbert, M.A., Eds.; Chapman and Hall: New York, 1992; 442–476.
3. Flanders, S.E. The bisexuality of uniparental hymenoptera, a function of the environment. *Am. Natural.* **1945**, *79*, 122–141.
4. Stouthamer, R.; Luck, R.F.; Hamilton, W.D. Antibiotics cause parthenogenetic *Trichogramma* to revert to sex. *Proc. Natl. Acad. Sci. U. S. A.* **1990**, *87*, 2424–2427.
5. Huigens, M.E.; Stouthamer, R. Parthenogenesis associated with *Wolbachia*. In *Insect Symbiosis*; Bourtzis, K.,

- Miller, T.A., Eds.; CRC Press: Boca Raton, FL, 2003; 247–266.
6. Zchori-Fein, E.; Gottlieb, Y.; Kelly, S.E.; Brown, J.K.; Wilson, J.M.; Karr, T.L.; Hunter, M.S. A newly discovered bacterium associated with parthenogenesis and a change in host selection behavior in parasitoid wasps. *Proc. Natl. Acad. Sci. U. S. A.* **2001**, *98*, 12,555–12,560.
 7. Grenier, S.; Pintureau, B.; Heddi, A.; Lassabliere, F.; Jager, C.; Louis, C.; Khatchadourian, C. Successful horizontal transfer of *Wolbachia* symbionts between *Trichogramma* wasps. *Proc. R. Soc. Lond., B* **1998**, *265*, 1441–1445.
 8. Huigens, M.E.; Luck, R.F.; Klaassen, R.G.H.; Maas, F.M.P.M.; Timmermans, M.J.T.N.; Stouthamer, R. Infectious parthenogenesis. *Nature* **2000**, *405*, 178–179.
 9. Timberlake, P.H.; Clausen, C.P. The parasites of *Pseudococcus maritimus* (Ehrhorn) in California. *Univ. Calif. Publ. Tech. Bull. Ent.* **1924**, *3*, 223–292.
 10. Silva, I.M.M.S.; Meer, M.M.M.v.; Roskam, M.M.; Hoogenboom, A.; Gort, G.; Stouthamer, R. Biological control potential of *Wolbachia*-infected versus uninfected wasps: laboratory and greenhouse evaluation of *Trichogramma cordubensis* and *T. deion* strains. *Biocontrol. Sci. Technol.* **2000**, *10*, 223–238.

Vegetable Crop Pest Management: Insects and Mites

Mark G. Wright

Plant and Environmental Protection Sciences, University of Hawaii at Manoa, Honolulu, Hawaii, U.S.A.

Mike Hoffmann

Department of Entomology, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

Vegetable crops are attacked by a wide variety of arthropod pests.^[1,2] They cause economic losses by feeding directly on the plants, either by devouring the tissue, or by piercing the tissue to feed on sap. In addition to causing physical damage to produce, some species transmit viruses and other microorganisms, which result in plant disease. Some of these diseases are extremely injurious, such as viral diseases of potatoes, tomato spotted wilt virus and yellow-top virus on tomatoes, and various viruses on cucurbits.

Management of arthropod pests in vegetable crops is often pesticide-intensive, owing to high quality requirements that markets set, the ephemeral nature of most vegetable crops, and the need to maximize profit margins. High quality requirements, often of an aesthetic nature, demand that produce be free of blemishes, scars, and foreign organisms. In addition, because most vegetables are annual crops, the effectiveness of classical biological control is reduced in some cases, as establishment of biological control agents may be tenuous. As a result of dependence on pesticides, numerous problems have arisen, such as pesticide resistance, resurgence of secondary pests, and nontarget effects of chemicals.^[3]

In response to the problems associated with the overuse of pesticides, integrated pest management (IPM) strategies are being developed and deployed for vegetables worldwide. Integrated pest management is a sustainable strategy that uses mutually compatible multiple tactics to suppress pests below economically important levels.^[3] The tactics include biological, cultural, mechanical, and chemical controls; the latter is only used when necessary. The use of IPM for control of arthropod pests in vegetables varies depending on the needs and the motivating factors of different farmers.

VEGETABLE PESTS AND MANAGEMENT

Vegetables contribute an important dietary component worldwide, providing variety and important nutritional

components (Table 1). Substantial areas, particularly in Asia and Europe, are planted to vegetable crops worldwide. Thus, vegetable farming has a potentially large impact on the environment and society. The need for safe yet effective pest management is therefore clear. Pesticide sales per continent (Table 1) provide a rough indication of the costs of controlling pest on vegetable crops.

Some pests have a worldwide distribution, such as certain thrips (Thysanoptera: *Thrips tabaci*, *Frankliniella occidentalis*), *Helicoverpa* spp. (Lepidoptera), potato tuberworm (Lepidoptera: *Phthorimaea operculella*), and red spider mite (Acari: *Tetranychus urticae*). Some of these attack a wide range of vegetable crops. For example, western flower thrips (*F. occidentalis*) has a broad food plant range, which includes most vegetable crops. In addition to causing physical damage on crops such as Cucurbitaceae, *F. occidentalis* also vectors tomato spotted wilt virus (TSWV), a devastating disease of Solanaceae and other crops. The relatively recent spread of *F. occidentalis* has been blamed for the intensification of TSWV in many parts of the world. The control of *F. occidentalis* is complicated by varying levels of insecticide resistance. In many cases, a range of pesticides are sprayed frequently or rotated to suppress *F. occidentalis* populations, often with limited success. Alternatives to pesticides include the use of cultivars resistant to TSWV (specifically tomatoes) and biological control of the thrips. Biological control relies on predatory mites (*Amblyseius cucumeris*) and minute pirate bugs (*Orius* spp.), but is generally inadequate for preventing spread of TSWV. The use of insect pathogens (such as *Verticillium lecanii*, *Beauveria bassiana*) may be a more promising alternative.

Another widespread pest that causes extensive damage to crops is the potato tuber moth (*P. operculella*). The larvae of this moth infest tubers in the ground as well as in storage. Yield loss can be high (23%), but usually averages 5%. Pesticides are used extensively to control potato tuber moth; however, IPM efforts are reducing this dependence. In Tunisia, for example, farmers are encouraged to use cultural techniques

Table 1 Annual worldwide production, consumption and cost of pesticides for vegetables, and predominant pest management practices

| | North and Central | | | | | |
|------------------------------|-------------------|-------------------|---------------|---------------|---------------|---------------|
| | Africa | Oceania/Australia | Europe | America | South America | Asia |
| Metric tons ^a | 186,819 | 8,153 | 238,047 | 104,790 | 73,857 | 753,909 |
| Hectares ^a | 72,738 | 3,449 | 100,989 | 47,790 | 21,152 | 422,842 |
| Consumption ^b | 36.08 (30.39) | 61.83 (45.25) | 98.45 (36.18) | 108.90 (2.83) | 42.72 (20.89) | 80.61 (60.38) |
| Pesticide sales ^c | 51,503 | 8,930 | 143,406 | 43,522 | 66,649 | 155,599 |
| Pest management | E;C;P | C;I;B | B;I;C | C;I;B | C;B;E | C;B;I |

C = chemical control; B = biological control; E = cultural control; P = physical control; I = IPM.

^a×1000.

^bAverage supply per capita per country, kg (±SD).

^cUS\$1000.

(From Ref.^[4].)

including timely harvest, irrigation to prevent soil cracks, and hilling in addition to insecticides. The use of IPM in Tunisia has resulted in savings of \$165,000 per year, a significant saving for a small country. Similar IPM practices are applied in many other countries. Other efforts in North Africa include the use of biological insecticides such as *Bacillus thuringiensis* and baculoviruses in storage, causing a shift away from the use of pesticides such as fenitrothion. Transgenic *Bt* potatoes are being tested for potato tuber moth control in New Zealand. There are also parasitic wasps that attack the larvae, and are reputed to be able to persist in fields in spite of pesticide applications.

Other pest species have comparatively restricted distributions or have a more restricted host crop range, yet cause massive crop losses. Several species of sweet potato weevil, for example, attack sweet potatoes, causing severe losses to tubers and vines. Sweet potato is considered to be the world's seventh most important food crop. It has been estimated that about \$300 per hectare may be lost to sweet potato weevils in the Dominican Republic. Although sweet potato weevils are widespread, the various species are restricted to different continents (Table 2).

Control of this pest is difficult, owing to the fact that larvae burrow into tubers. Insecticidal control is most commonly applied as a soil drench or granules at planting. Some foliar applications of insecticides may be made to reduce adult numbers. Integrated pest management shows greater promise than relying on pesticides, and has been shown to provide increased yields. IPM options include cultural, biological, and physical control measures. The most important cultural practices are ensuring isolation of plantings, sanitation, deep plowing, hilling around tubers, deep planting, or the use pseudo-resistant, deep developing cultivars. The removal of alternative weed hosts such as morning glory (*Ipomoea pandurata*) is an important component of cultural control. Biological control using entomopathogenic nematodes (*Steinernema*

carpocapsae and *Heterorhabditis bacteriophora*) is being investigated in the United States. Mass trapping adults with pheromone traps appears to reduce sweet potato weevil numbers in the United States and Asia. Up to 10% reductions in damage to foliage and 58% for tubers has been attributed to the use of trapping. Pest densities can also be monitored using pheromone traps.

Biological control of insect pests has been used since the 12th century in China, where predatory insect populations were augmented by releases of indigenous insects. Chinese pest management subsequently went through a period of reliance on pesticides, but this was largely replaced by the implementation of integrated pest management for a period. Implementation of IPM in China during this time was facilitated to a large extent by land-division practices there. Land was divided into small portions and divided between farmers in an area. Each was then able to plant the crops of his choice. This resulted in a complex mosaic of crops, encouraging beneficial insects. Pest monitoring was also done actively by Chinese farmers or regional pest scouts, further enhancing application of IPM. Another great advantage that Chinese farmers have is indigenous knowledge of effective pest management practices dating back to the 12th century. Some old but effective practices are still applied today. However, more recent developments in China have resulted in many farmers reverting to the use of pesticides.

Table 2 Sweet potato weevil species from different parts of the world

| Continent | Weevil species |
|--------------------|-----------------------------------|
| Africa | <i>Cylas puncticollis</i> |
| Sub-Saharan Africa | <i>C. brunneus</i> |
| Asia | <i>C. formicarius</i> |
| North America | <i>C. formicarius elegantulus</i> |
| South America | <i>Eusepeles postfasciatus</i> |

In only a few instances is the natural biological control of a pest incorporated directly into the IPM program. In California, U.S.A., the tomato fruitworm (*Helicoverpa zea*) is the principal pest of processing tomatoes. Over 90% of U.S. processing tomatoes are grown in California. Fields are monitored for tomato fruitworm eggs to determine if an insecticide treatment is warranted. However, the treatment threshold is adjusted to account for egg parasitism by naturally occurring *Trichogramma*, based on the ratio of white to black (parasitized) eggs recorded while fields are monitored. This technique is used on about 30% of the crop in some areas of the state. Research on biological control of pests is ongoing in various parts of the world, with the potential to provide considerable benefits.^[5]

Motives for the application of IPM to vegetable crops vary between continents and cultures. In many cases, the decision to apply IPM is an economic one, albeit for contrasting reasons in different socio-economic climates. In first-world countries, IPM may be applied as a response to market demands, while in less developed countries, IPM is the default effect of not being able to afford pesticides and the equipment necessary to apply them. Subsistence farmers in many cultures apply the principles of IPM incidentally, simply to ensure that they are able to harvest yields that will at least fulfill their household needs. Their practices include planting polycultures, physical control of pests, and planting times. Pesticides are absent from their arsenal of alternatives in many cases. More affluent farmers tend to plant larger monocultures, with concomitant increased dependence on pesticides. Subsistence farmers tend to be concentrated in tropical and subtropical parts of Africa,^[6] parts of Asia, South America, and Central America. Most of Europe, North America, Australia, and parts of Asia are dominated by large-scale commercial farming, with higher inputs.

Conventional broad spectrum insecticides such as the organophosphates and carbamates are widely used in vegetables. In recent years, entirely new classes of insecticides have become available. These new compounds are more selective and generally less toxic to non-target organisms. Several compounds such as macrocyclic lactones and pyrroles (e.g., spinosad, emamectin benzoate, chlorofenapyr) are several times more effective per unit toxin than most other conventional insecticides. Consequently, they can be applied at very low rates. They also do not show cross-resistance with the existing insecticides on the market.

Botanical pesticides provide another alternative to synthetic pesticides in vegetable pest management. Neem extract, *Azadirachta indica* (Meliaceae), is particularly renowned for its insecticidal properties,

and has been used as an insecticide for many years in India as a crude aqueous extract. Neem has relatively recently been recognized for its potential in the Western world, and commercial formulations and analogues of azadarachtin have been produced. Neem extract and other botanical pesticides can play a valuable role in pest management on vegetables, where resistance is often a factor, providing unique chemistry for inclusion in alternation programs.

Other pesticides of natural origin, such as *B. thuringiensis* (*Bt*) products, are also used in vegetable pest management. *Bt* products are environmentally safe (relative to other pesticides), and are even acceptable for organic vegetable production. Despite the desirable characteristics of *Bt* products, they constitute less than 2% of all insecticides. A relatively new use of *Bt* is in transgenic crops incorporating *Bt* genes. This approach appears very attractive, but remains controversial because of adverse public reactions.

In general, within the agrochemical field, there is a trend toward the use of more selective pesticides and formulations that reduce application rates. Pesticide companies are also aware of the need to implement IPM, not only for environmental reasons, but also for the management and prevention of resistance to products. In various countries, there are regulatory considerations influencing pesticide use. In the United States, the Food Quality Protection Act has important implications, as registrations for many insecticides will be withdrawn on various vegetable crops. In Europe, some countries, such as Denmark, have strict legislation enforcing the reduction of pesticide use. These measures force vegetable farmers to move toward the use of integrated pest management.

REFERENCES

1. McKinlay, R.G., Ed.; *Vegetable Crop Pests*; CRC Press Inc.: Boston, Massachusetts, 1992.
2. Hoffmann, M.P.; Petzold, C.H.; Frodsham, A.C. Integrated Pest Management for onions. In *Cornell Cooperative Extension, NY State IPM Program Publication No. 119*; Cornell University: Ithaca, New York, 1996.
3. Pedigo, L.P. *Entomology and Pest Management*, 3rd Ed.; Prentice-Hall Inc.: Upper Saddle River, New Jersey, 1996.
4. FAOSTAT—Agriculture Data. 1999. <http://apps.fao.org/cgi-bin/nph-db.pl?subset=agriculture> (accessed March 2002).
5. Waterhouse, D.F. *Biological Control of Insect Pests: Southeast Asia Prospects*; ACIAR Monograph No. 51: Canberra, 1998.
6. Kumar, R. *Insect Pest Control with Special Reference to African Agriculture*; Edward Arnold Publishers Ltd.: London, 1984.

Virus for the Biological Control of Insects

Marlinda Lobo de Souza

Maria Elita Batista de Castro

Embrapa Recursos Genéticos e Biotecnologia, Brasília, Brazil

Flavio Moscardi

Embrapa Soja, Londrina, Brazil

INTRODUCTION

Biological control using microorganisms is a very promising and fascinating field of research. Insect viruses can be applied as agents for the control of pests and disease vectors. Among them, baculoviruses comprise the most important group for biocontrol purposes. The use of baculoviruses as alternative to chemical insecticides is mainly attributable to their features, such as those relating to safety on human health and the environment. Knowledge about the biology of these viruses and their application as bioinsecticides has increased in recent decades. However, some factors must be considered for the success of biological control programs: viral specificity, insect behavior, tolerance of the crop to damage, and costs of mass production. Furthermore, some limitations remain for in vitro commercial production such as availability of cell lines and molecular changes, with loss of virulence, caused by the serial passage of the virus in cell culture.

VIRUSES ASSOCIATED TO INSECTS

The following families of invertebrate viruses are currently classified on the Seventh Report of the International Committee on Taxonomy of Viruses:^[1] *Poxviridae*, *Iridoviridae*, *Baculoviridae*, *Polydnaviridae*, *Ascoviridae*, *Circoviridae*, *Parvoviridae*, *Reoviridae*, *Birnaviridae*, *Rhabdoviridae*, *Bunyaviridae*, *Picornaviridae*, Cricket paralysis-like viruses, *Tetraviridae*, *Nodaviridae*, *Togaviridae*, *Flaviviridae*, and *Metaviridae*. The *Baculoviridae* is a large family of occluded viruses composed of two genera [*Nucleopolyhedrovirus* (NPV) and *Granulovirus* (GV)], which are differentiated by the size and form of their occlusion. Baculoviruses are efficient, highly specific, safe to vertebrates, beneficial arthropods, plants, and microorganisms, and can be integrated with other control tactics in a pest management program. For these reasons, baculoviruses have been the most researched group of insect viruses toward their application in pest control (Fig. 1).

PROGRAMS USING VIRAL PESTICIDES

In the last two decades, baculoviruses have been used commercially to control a number of pests, including: *Anticarsia gemmatilis* (AgMNPV) in Brazil; *Cydia pomonella* (CpGV), *Spodoptera exigua* (SeMNPV), *Trichoplusia ni* (TnMNPV), *Helicoverpa zea*, and *Heliothis virescens* (both with HzSNPV) in the USA; *Spodoptera exigua* (SeMNPV) and *Cydia pomonella* (CpGV) in Europe; *Helicoverpa armigera* in China, India, and Australia; *Spodoptera litura* in China; and forest pests as *Lymantria dispar* (LdMNPV), *Orgyia pseudotsugata* (OpMNPV), *Choristoneura fumiferana* (CfMNPV), and *Neodiprion sertifer* (NeseNPV) in different countries. The most successful example of a program using a viral pesticide is the use of the *Anticarsia gemmatilis* MNPV to control the velvetbean caterpillar in soybean. This program was implemented in Brazil in the early 1980s, and currently over 1,700,000 ha of soybean are treated annually with this virus.^[2] For a detailed account on the different programs worldwide, see Ref.^[2] and references therein (Fig. 2).

BACULOVIRUS PATHOGENESIS

Baculoviruses are distinguished by the production of two structurally and functionally distinct virion phenotypes in their life cycle, the occluded virus (OV) and budded virus (BV). Infection is initiated after host larvae ingest occluded virus particles [called polyhedra or granules or occlusion bodies (OBs)], which are dissolved through the action of the alkaline digestive juices of the insect midgut. Dissolution of OBs is aided by both the high pH, characteristic of most Lepidoptera larvae, and the presence of proteases in the insect gut. Infection is first observed in the epithelial cells of the midgut, and this is followed in most cases by a systemic infection after the virus passes through the basal lamina and reaches the larval hemocoel. The BV phenotype is produced by budding from surfaces of infected cells and serves to transmit the virus among



Fig. 1 Velvetbean caterpillar infected with baculovirus (*Anticarsia gemmatalis* nucleopolyhedrovirus—AgMNPV).

various cells and tissues of the insect. Late in infection, occluded viruses are embedded in a crystalline matrix in the nucleus to form occlusion bodies that are highly stable in the environment and are responsible for the horizontal transmission of the disease. After ingestion of the virus, the host larvae are debilitated, resulting in reduction of development, feeding, and mobility followed by massive tissue infection and release of numerous OBs into the environment upon the death of the insect host, thereby completing the infection cycle.^[3]

The occlusion-derived viruses (ODV), which are found within the occlusion bodies, are highly infectious to epithelial cells of insect midgut and enter cells by fusion of the virion envelope with the plasma membrane at the cell surface. However, adsorptive endocytosis is the major pathway for baculovirus BV infection.^[4] Because BVs are non-occluded in a protective form, they are not infectious to larvae by ingestion. Nevertheless, BVs are highly infectious to cultured cells and to tissues when injected in the larval hemocoel.^[5,6]

CHARACTERIZATION OF INSECT VIRUSES

The identification and characterization of insect viruses are essential for finding new bioinsecticides, for registration of commercial products, and for acquiring more information about their relatedness and phylogeny. The establishment of insect cell lines supporting the replication of baculoviruses resulted in purification of viral clones through plaque assays and promoted the advance of the baculovirus molecular biology. Most of the studies were performed with *Autographa californica* MNPV because of its easy propagation and relative stability in cell culture, and also because of its relatively broad host range.

Several techniques have been routinely used for characterization of insect virus. Most of them initially require the isolation of viral particles from insect tissues and debris.^[7] The purification process is based on differential centrifugation, which consists of periods of speed of sedimentation, high and low, allowing the separation of viruses from other material. Centrifugation in sucrose gradient is applied when a higher degree of purification of the viral particles is required. Once purified, these particles can be characterized at morphological, biochemical, and molecular levels (Fig. 3).

Transmission electron microscopy and light microscopy are usually used as a first step to virus identification. Other common techniques include protein analysis by sodium dodecyl sulfate-polyacrylamide gel electrophoresis (SDS-PAGE), serological identity/relatedness using immunodiffusion, enzyme-linked immunosorbent assay (ELISA), and Western blots. However, the most widely technique used to characterize viruses is the restriction enzyme analysis of viral DNA (REN). This methodology became important in distinguishing



Fig. 2 Production and formulation of the bioinsecticide *Baculovirus anticarsia*.

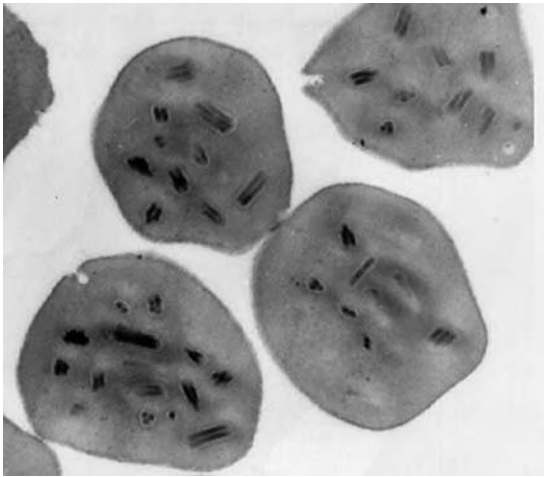


Fig. 3 Electron micrograph of AgMNPV polyhedra.

viral isolates and for the construction of physical maps of the viruses.

In the early 1990s, most of the studies on phylogeny of baculoviruses included techniques of digestion with restriction enzymes, DNA hybridization, and comparison of amino acids and nucleotide sequences, transcriptional maps, and functional analysis of genes.^[8] Nowadays, other molecular techniques such as cloning, sequencing, and PCR analysis are increasingly used in many laboratories. Currently, the genomes of more than 20 baculoviruses have been completely sequenced, a development that has allowed important phylogenetic studies into the virus group and their relationship with the host.

ANALYSIS OF VIRAL POPULATIONS

Wild-type populations of baculoviruses have been studied via comparison of the DNA restriction profiles of the viral isolates. Geographical isolates are those of the same virus collected in different regions, whereas seasonal isolates are collected from the same area, but in different seasons, where the virus was applied, usually in a biocontrol program. Genotypic variants can occur among both geographical and seasonal isolates. The occurrence of these variants in the wild-type isolates of baculoviruses populations is well documented. They refer to baculoviruses with very similar genomes that can be distinguished by differences in the restriction profile. Such variants are easily recognized by the presence of submolar fragments of DNA present in the profiles.^[9] Recombination among the natural isolates is one of the events that can lead to the formation of these variants. This is an important mechanism to increase diversity in viral populations (Fig. 4).

Another important parameter is the measurement of the biological activity of the virus, which is important in the selection of highly pathogenic viral isolates and the monitoring of the quality of a viral insecticide used in a biocontrol program. Bioassays are procedures commonly used to evaluate the pathogenicity of the virus in its insect host or to investigate its host range.^[10] Viral pathogenicity is mainly characterized by measurement of its mean lethal dose (LD₅₀) and the mean lethal time (LT₅₀).

VIRUS COMMERCIAL PRODUCTION

The host specificity of baculoviruses is desirable in integrated pest management programs, but the potential market is restricted, as are economic returns, a factor that might have influenced the decision by some private companies to discontinue development or sales of some viral insecticides. Another limiting factor for industry is the large-scale production of baculoviruses. Currently, this can be accomplished only *in vivo*, mostly on insects reared in artificial diets and, in some cases, under field conditions. Production using laboratory-reared insects has progressed technically and economically, but the process is still too costly to render most baculoviruses to be cost-competitive with chemicals. Another important factor that limits industry interest and farmer acceptance of baculoviruses is their slow rate in stopping pest damage and killing the hosts. To overcome these limitations, some strategies have been proposed such as the use of substances

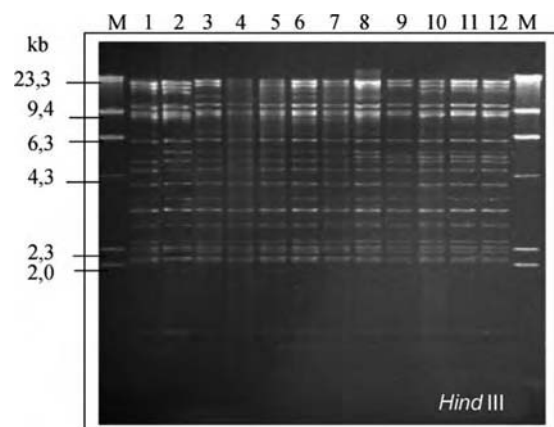


Fig. 4 Analysis of the DNA of seasonal isolates of AgMNPV collected in the same area, in Londrina, Brazil, during successive years of virus application. The viral DNA was digested with the *Hind*III enzyme and analyzed by agarose electrophoresis: molecular weight marker (M); AgMNPV original viral isolate^[11] and isolates from seasons 1984/1985,^[2] 1985/1986,^[3] 1986/1987,^[4] 1987/1988,^[5] 1989/1990,^[6] 1990/1991,^[7] 1992/1993,^[8] 1993/1994,^[9] 1994/1995,^[10] 1995/1996,^[11] and 1996/1997.^[12]

enhancing baculovirus activity in host insects (boric acid, chitinase, optical brighteners) and genetic engineering of virus by reducing time to kill host insects and their feeding capacity.

Genetic modification of the baculoviruses became possible after the development of baculovirus expression vector system technology.^[11] The natural insecticidal activity of these viruses can be improved by inserting foreign genes encoding insect-specific toxins (e.g., scorpion and mite venoms), hormones, enzymes, or other gene products exhibiting insecticidal activity. Improvement of the speed of action can also be achieved by deletion of viral genes, such the ecdysteroid glucosyl-transferase gene (*egt*), which encodes an enzyme that inactivates ecdysteroid hormones.

The commercial production of baculoviruses in insect cells (in bioreactors) is still not feasible technically and economically. The main problems are high costs, availability of susceptible cell lines, and molecular changes resulting from serial passage of the virus. Defective interfering particles (DIP) and few polyhedra mutants (FP) are the most common mutations caused by the passage effect.^[12] They usually lead to loss of virulence for the target insect. However, appropriate strategies for the production in bioreactors and the greatest amount of information generated from the virus molecular biology will certainly contribute to overcome these difficulties in the near future.

CONCLUSION

Despite their potential capability to control pests worldwide, baculoviruses have so far been unable to fulfill their promise to control pests in crops, forests, and grasslands. The most important developments were made with the NPV of *A. gemmatalis* in soybean in Brazil (area wide), the NPV of the *Helicoverpa/Heliiothis* complex (United States, China, India, and Australia), NPVs of the *Spodoptera* complex (*S. litura* and *S. exigua*) in Europe and Asia, the NPV of *L. dispar* in many countries, and the GV of *C. pomonella* in the United States and Europe. These programs show that the use of viral insecticides based on baculoviruses is a viable alternative to chemical insecticides. However, expansion of their use will depend on research or actions on key limiting issues, such as solar radiation, host specificity, slow rate to kill insects, and passage effect in cell culture. Furthermore, development

of new strategies to improve viral mass production and to render baculoviruses cost-competitive with chemicals will be essential for their commercialization and widespread use as biopesticide.

REFERENCES

1. Van Regenmortel, M.H.V.; Fauquet, C.M.; Bishop, D.H.L.; Cartens, E.B.; Estes, M.K.; Lemon, S.M.; Maniloff, J.; Mayo, M.A.; McGeoch, D.J.; Pringle, C.R.; Wickner, R.B. Virus taxonomy. Classification and nomenclature of viruses. In *Seventh Report of International Committee on Taxonomy of Viruses*; Academic Press: San Diego, CA, 2000.
2. Moscardi, F. Assessment of the application of baculoviruses for the control of Lepidoptera. *Annu. Rev. Entomol.* **1999**, *44*, 257–289.
3. Federici, B.A. Baculovirus pathogenesis. In *The Baculoviruses*; Miller, L.K., Ed.; Plenum Press: New York, 1997; 33–59.
4. Blissard, G.W. Baculovirus—Insect cell interactions. *Cytotechnology* **1996**, *20*, 73–93.
5. Volkman, L.E.; Summers, M.D. *Autographa californica* nuclear polyhedrosis virus: comparative infectivity of the occluded, alkali-liberated, and nonoccluded forms. *J. Invertebr. Pathol.* **1977**, *30*, 102–103.
6. Keddie, A.; Volkman, L.E. Infectivity difference between the two phenotypes of *Autographa californica* nuclear polyhedrosis virus: importance of the 64K envelope glycoprotein. *J. Gen. Virol.* **1985**, *66*, 1195–1200.
7. Tompkins, G.J. Purification of invertebrate viruses. In *Atlas of Invertebrate Viruses*; Adams, J.R., Bonami, J.R., Eds.; CRC Press: Boca Raton, 1991; 31–40.
8. Possee, R.D.; Rohrmann, G.F. Baculovirus genome organization and evolution. In *The Baculoviruses*; Miller, L.K., Ed.; Plenum Press: New York, 1997; 109–140.
9. McIntosh, A.H.; Rice, W.C.; Ignoffo, C.M. Genotypic variants in wild-type populations of baculoviruses. In *Biotechnology in Invertebrate Pathology and Cell Culture*; Maramorosch, K., Ed.; Academic Press: San Diego, 1987; 305–325.
10. Evans, H.; Shapiro, M. Viruses. In *Manual of Techniques in Insect Pathology*; Lacey, L., Ed.; Academic Press: San Diego, 1997; 17–53.
11. Treacy, M.F. Recombinant baculoviruses. In *Methods in Biotechnology*; Hall, F.R., Menn, J.J., Eds.; Biopesticides: Use and Delivery; Humana Press: Totowa, NJ, 1999; Vol. 5, 321–337.
12. Krell, P.J. Passage effect of virus infection in insect cells. *Cytotechnology* **1996**, *20*, 125–137.

Vole Management for Orchards

David Lockwood

*Department of Plant Sciences, University of Tennessee,
Knoxville, Tennessee, U.S.A.*

INTRODUCTION

Roots and stems of herbaceous plants make up the preferred food sources for voles. When these become limited, voles may feed on the bark of trees. Even at low populations, voles cause significant damage to orchards, nurseries, and landscapes throughout most parts of North America.

Prevention or reduction of vole damage in orchards entails a number of practices depending on the numbers of voles present. Monitoring orchards to discern vole populations should be a routine orchard management practice. The economic threshold of damage is very low because a single vole can seriously wound a tree.

DESCRIPTION OF VOLES AND HABITAT

Biology

Population levels peak about every 4 years; however, these cycles are not predictable.^[1] Food quality, climate, predation, physiological stress, and genetics can affect population levels. Under favorable conditions, vole populations may increase very quickly.

Behavior

Meadow voles and prairie voles are primarily surface feeders. They live and work in runways constructed in vegetation and litter on the soil surface. Damage from them will occur on tree trunks near the soil surface. Pine voles spend most of their time in underground tunnels. They girdle tree trunks or roots below ground level.^[2,3]

Damage

In the United States, annual crop losses in apple orchards due to vole damage were estimated to be \$50 million prior to the widespread use of rodenticides.^[4] A recent nationwide survey of orchardists revealed that approximately 123,000 apple trees were killed annually by voles.^[5]

In late fall and winter, preferred food sources become limiting. Voles will turn to the bark at the base of tree trunks and roots as an alternate food source.^[6,7]

Injured trees may be slow growing, look sickly, and become loose in the soil. Damage may not be readily apparent until it is extensive and recovery is unlikely, especially with pine vole damage.^[8]

MONITORING FOR THE PRESENCE OF VOLES

Orchards should be monitored in late fall when the population will be highest, to determine whether the use of a rodenticide is warranted, and in spring to assess the effectiveness of controls and the potential for vole buildup over the summer months. Several different types of monitoring may be employed.^[1]

Trapping

Trapping, while not an effective control practice, is valuable for checking on the presence of voles and for determining what type of vole exists in the orchard, thus enabling selection of the most effective types of control. Place traps in runways or tunnels in late afternoon. Bait traps with peanut butter or an apple slice. Check traps the following morning^[1] (Table 1).

Concentration Stations

Concentration stations should be placed on the sod near the dripline of trees throughout the orchard in the latter part of summer. After the station has been in place a couple of months, check under the station for the presence of tunnels or runways, which would signify an active vole population. Use about 10 stations per acre of orchard as the home range of a vole may be quite small.^[1,8]

Apple Sign Test

The apple sign test consists of placing a piece of wood or shingle over a hole or runway. After a week, a piece of apple about the size of a quarter should be placed in the runway or hole under the cover. Check the day after baiting to see if the apple has been partially consumed or is missing. To estimate the vole population, weigh the apple piece at the time it is put out and again

Table 1 Distinguishing characteristics of voles

| Characteristic | Meadow voles | Prairie voles | Pine voles |
|--------------------|--|--|---|
| Length (with tail) | 5½ to 7½ in. | 5 to 7 in. | 4 to 6 in. |
| Tail | At least twice the length of the hind foot | At least twice the length of the hind foot | Same length or shorter than the hind foot |
| Adult fur | Gray to yellow brown obscured by black-tipped hairs | Gray to dark brown, mixed with gray, yellow, or hazel-tipped hairs | Brown, soft, dense |
| Eyes | Large | Large | Small |
| Ears | Large | Large | Inconspicuous |
| Nest | Usually above ground, but occasionally in shallow burrows | Usually above ground, but occasionally in shallow burrows | In shallow burrows |
| Habitat | Old fields, ditch banks, pond banks, fence rows, orchards, pastures | Old fields, marshland, orchards | Old fields, thickets, orchards, edges of agricultural lands, especially where soils are loose and sandy |
| Food | Grasses, seeds, grain, bark, some insects | Grasses, seeds, grain, bark | Bulbs, tubers, seeds, bark |
| Damage | Girdling of tree trunks at or just below the groundline, shallow roots | Girdling of tree trunks at or just below the groundline, shallow roots | Girdling of tree trunks and roots |

Source: From Refs.^[1,6,9].

24 hr later. A pine vole will consume about 13 g of apple in 24 hr and a meadow vole will consume about 20 g.^[8]

The guard should extend from about 3 in. below the soil line to 18 in. above it. Guards will not protect against pine voles.^[2,9,10]

CONTROL

An integrated vole management program utilizing monitoring and several control options will give the best results.

Predation

Dogs, cats, hawks, owls, snakes, coyotes, and foxes will prey on voles. While their activity will not be sufficient to significantly reduce a high vole population, they may be able to hold low populations in check, especially in conjunction with other controls. Encourage the presence of predators by creating sites that favor them.^[2,8]

Repellents

Thiram and capsaicin are registered for use as repellents for voles. They should be applied to the base of trees as a spray or combined with white latex paint and applied to the trunks. Use of repellents should not be relied on exclusively.^[6]

Exclusion

Guards around the base of young trees can prevent damage from the surface-feeding voles and rabbits.

Habitat Modification

Habitat modification involves creating an environment in the orchard that does not favor vole presence or activity. Maintain a vegetation-free area extending at least 2 to 3 ft out from the base of trees plus frequent, close mowing of the vegetation in and around orchards. Crushed stone or sand against the base of trees creates an area that is difficult for voles to construct runways or tunnels. Shredding or removing dropped fruit and leaf litter following harvest will remove a food source and destroy an environment favorable for runway or tunnel construction. Shallow tillage will destroy runways and some tunnels and kill a percentage of the vole population.^[6] Cleaning up fence rows, ditch banks, and pond banks around orchards will discourage vole buildup near orchards. Good habitat modification will favor predation as voles will be more exposed.^[1]

Rodenticides

Rodenticides, when combined with habitat modification, provide the quickest way to reduce large vole populations. The two types of rodenticides used in orchards are acute toxicants (zinc phosphide) and

anticoagulants.^[8] An acute toxicant will provide a lethal dose of poison in one feeding, whereas anticoagulants depend on repeated consumption.

Rodenticides should be put out in late fall after preferred food sources are no longer available. Baits should not be placed on bare soil, as voles tend to avoid these areas. Pelleted formulations of baits are preferred to grain baits as they tolerate adverse weather conditions better and pose less of a threat to non-target wildlife. Select a period where no precipitation is expected for several days to put out baits.^[9]

Spot or trail baiting involves placement of the rodenticide in surface runways or at the mouths of holes leading to underground burrows. Avoid disturbing runways. Cover bait by pulling overhanging grass back in place. Bait should be placed in several spots near the base of infested trees.

Where concentration (bait) stations have been put in place earlier in the growing season, bait may be placed under them. The station will lessen the potential for exposure of non-target species to the bait and protect the bait during adverse weather conditions. Boards, shingles, or metal pieces measuring at least 15 in. by 15 in. make good stations. Car tires split horizontally and placed with the hollow side down and inverted T's made from 1½-in. ABS pipe also make good stations.^[8,11]

Monitor orchards to determine the level of residual populations or to detect vole movement into the area. Reapplication of rodenticides may be needed.

Be sure to check to see which rodenticides are legal for use in your area. Always read and follow label directions concerning their use.

CONCLUSION

Voles can cause serious damage to fruit trees in many years. Vole populations should be monitored annually to determine which control measures are warranted. Successful vole control involves an integrated

management approach. Refer to recommendations for your state when considering the use of rodenticide.

REFERENCES

1. Lockwood, D. Vertebrate management. In *Integrated Orchard Management Guide for Commercial Apples in the Southeast, AG-572*; Walgenbach, J., Ed.; North Carolina State University: Raleigh, NC, 2003; 65–69.
2. Thurston, S.N.; Brittingham, M.C.; Williams-Whitmer, L.M. *Voles. Wildlife Damage Control 9*. <http://www.cas.psu.edu> (accessed August 2003).
3. Johnson, M.L.; Johnson, S. Voles. In *Wild Animals of North America: Biology, Management and Economics*; Chapman, J.A., Feldhammer, G.A., Eds.; The Johns Hopkins University Press: Baltimore, MD, 1982; 326–354.
4. Byers, R.E. Control and management of vertebrate pests in deciduous orchards of the eastern United States. Chapter 7. *Hortic. Rev.* **1984**, *6*, 253–285.
5. Curtis, P.D.; Fargione, M.J.; Richmond, M.E. Wildlife damage management in fruit orchards. *Cornell Coop. Ext. Inf. Bull.* **1994**, *236*, 10.
6. *Voles: Identification, Habitat, Damage, Prevention, Control*. http://www.pestproducts.com/voles_meadow_mice.htm (accessed May 2003).
7. *Voles in Commercial Orchards and Ornamental Nurseries, Wildlife Damage Management Fact Sheet AG-472-1*. <http://www.ces.ncsu.edu/nreos/wild/wildlife/wdc/voles.html> (accessed August 2003).
8. Brittingham, M.C.; Williams, L. Mammal control in orchards. In *Pennsylvania Tree Fruit Production Guide, 1998–1999*; Pennsylvania State University: University Park, PA, 1998; 122–124.
9. Tobin, M.E.; Richmond, M.E. *Meadow Vole and Pine Vole. Tree Fruit Fact Sheet 102GFSTF-M1*; 1988. <http://www.nysipm.cornell.edu/factsheets/treefruit/pests/vole/voles.html> (accessed August 2003).
10. Kays, J.; Dutky, E. *Wildlife Damage Management: Reducing Vole Damage to Plants FS-654*. <http://www.agnr.umd.edu/MCE/Publications/Publication.cfm?ID=146&cat=8> (accessed June 2003).
11. Wilson, K.R. *Rodent and Deer Control in Orchards*. <http://www.gov.on.ca/OMAFRA/english/crops/facts/98-023.htm> (accessed March 2).

Weed Management: Biological and Chemical Approaches

Thomas W. Culliney

Center for Plant Health Science and Technology, USDA, APHIS, PPQ,
Raleigh, North Carolina, U.S.A.

INTRODUCTION

Weed management combines the elements of prevention, eradication, and control to eliminate or mitigate weed problems. Central to the concept is an understanding of weed biology to discover the causes of weed problems. A good weed management program emphasizes prevention over control. If prevention fails, control measures must be employed to minimize the economic impact of a weed infestation. Two major approaches to weed control involve the application of biological methods and chemical compounds.

BIOLOGICAL CONTROL

Classical biological control, involving the importation, colonization, and establishment of exotic natural enemies (predators, parasites, and pathogens) to reduce pest populations to, and maintain them at, densities that are economically insignificant, is the predominant method employed in biological weed control.^[1] Inoculative release, whereby natural enemies are liberated once or over a limited period of time to establish self-perpetuating populations, is the mainstay of biological weed control. Conservation, the manipulation of the environment to favor (often native) natural enemies, is rarely employed. Augmentation, the mass production and periodic colonization of enemies, usually involves mycoherbicides (fungi applied in inundative doses like a chemical herbicide) and some insects, but also the use of grazing animals, to control weeds.

Natural enemies may control weeds directly, by destroying vital parts, leading to the death of the plant; or, indirectly, by making the weed more susceptible to attack from pathogenic or saprophytic organisms, or by exerting sufficient stress on the weed so as to put it at a competitive disadvantage to other valued plants. To be effective, natural enemies must respond in a density-dependent manner to changes in the target weed's population, thus serving to regulate the weed's abundance.

Implementation of biological weed control programs requires many years of research, much of them devoted to the exhaustive host-range testing necessary to ensure that any biotic agent released will be highly specific to the target weed, and often involves international cooperation. Classical biological weed control

programs all have in common a number of procedures (Table 1).

Biological control has been used primarily against weeds in aquatic, pasture, and rangeland habitats. Targeted weeds often have been economically or environmentally important and not amenable to other controls, or their ranges have expanded to such an extent that control by other available methods was not considered economically feasible. Indeed, the more widespread and damaging a weed is, the greater the benefits of biological control are.^[1] The first successful case of biological weed control occurred in 1836, when the mealybug *Dactylopius ceylonicus* (Green), native to Brazil, was used in India to control the introduced cactus *Opuntia vulgaris* Miller.^[2] In the United States, biological weed control had its beginnings in Hawaii in 1902, with the introduction of 23 species of insects from Mexico for the control of *Lantana camara* L. (Verbenaceae), a pest of lowland pasture. Countries most active in classical biological control of weeds are the United States, Australia, South Africa, Canada, and New Zealand; other countries involved to varying degrees include Malaysia, Thailand, India, Indonesia, Vietnam, Papua New Guinea, and China. Over the past 100 years, more than 350 organisms, including arthropods (insects and mites), fungi, and, to a lesser extent, nematodes and vertebrates (fish, grazing mammals, and fowl), have been released for the biological control of weeds worldwide.^[3] A total of 949 releases was made from the late 19th century to the end of 1996 for the control of at least 212 weed species. During the past two decades, there has been a steady increase in the number of weeds targeted for biological control, from 82 in 1982 to 133 in 1998. There also has been an increase with time in the success rate (slightly better than 50%), although determination of success presents difficulties, as it often has relied on subjective assessments. Biological weed control programs also have had a laudable safety record, with only eight cases (2%) worldwide in which non-target plants were attacked by introduced natural enemies, and in none of these cases was serious economic or environmental damage caused.^[3]

Biological control has recognized drawbacks. It is slow-acting compared with mechanical or chemical means of control, often less immediately effective and certain in its outcome, and is ineffective in controlling the weed *complex* present in many cropping systems.^[4]

Table 1 General procedures followed in classical biological weed control programs

| |
|---|
| Preliminary evaluation of the ecology and economic impact of the target weed |
| Survey of natural enemies already attacking the weed in the new habitat to reveal accidentally introduced agents and thus eliminate them from future evaluation |
| Literature search, survey, and identification of agents attacking the weed and its close relatives in their native regions |
| Screening of candidate agents in the foreign country to determine host range and specificity, and to eliminate non-specific agents from further consideration |
| Further testing of promising candidates in quarantine after introduction |
| Release of host-specific agents |
| Postrelease evaluation to determine establishment and effectiveness of agents |
| Redistribution of agents to other areas where control is needed |

However, when implemented properly, biological control can yield permanent, cost-effective management of weed populations with minimal environmental disturbance. In some cases, the economic returns have been spectacular (Table 2), in part owing to the fact that proven agents can be redistributed to new countries at very little cost. Biological control often is the only feasible means of controlling weeds infesting ecologically fragile conservation areas.

CHEMICAL CONTROL

Probably for as long as there have been weeds to vex mankind, toxic chemicals have been used to control

them. Readily available, natural substances, such as salt (sodium chloride), mineral oils, plant extracts, lime, and wood ashes, were used for centuries for weed control. Simple inorganic compounds, such as sulfuric acid, iron sulfate, copper sulfate, sodium chlorate, sodium borate, and sodium arsenite, came into use after 1821.^[4,5] Dinitrophenol, the first synthetic organic herbicide for selective weed control, was introduced in France in 1932. However, the modern era of chemical weed control was ushered in with the synthesis, in 1941, of the plant growth regulator 2,4-D. Development of new herbicides accelerated after World War II. Today, synthetic herbicides are dominant in weed management programs in the developed world, constituting 47% of world agrochemical sales as compared with insecticides, which comprise only 29%.^[1] The United States accounts for fully one third of the global market for herbicides. Over 85% of herbicides are used in agriculture.

Herbicides may be classified according to time of application or mode of action.^[4] Application times, according to stage of the crop, include: preplanting—the herbicide is applied to weed foliage before planting or is incorporated into the soil; preemergence—the herbicide is applied either to the soil surface or is incorporated into the soil after planting, but before emergence of the crop; emergence—application is made as the crop is emerging from the soil; and postemergence—the herbicide is applied, either as a broadcast or as a directed spray, after the crop is well established. According to mode of action, herbicides fall into eight main groups: 1) photosynthesis inhibitors, which cause a gradual chlorosis in plants; 2) pigment production inhibitors, which disrupt carotenoid synthesis or inhibit protoporphyrinogen oxidase, an enzyme essential to chlorophyll synthesis; 3) lipid biosynthesis inhibitors, which destroy plant structure by acting on membranes or cuticular waxes; 4) amino acid biosynthesis inhibitors, which attack enzymes essential for the synthesis

Table 2 Economic benefits of some biological weed control programs

| Target weed | Country | Return on investment (%) | References |
|---|---------------|--------------------------|------------|
| <i>Alternanthera philoxeroides</i> (Martius) Grisebach | United States | 610 ^a | [8] |
| <i>Centaurea</i> spp. | Canada | 19 ^a | [5] |
| <i>Chondrilla juncea</i> L. | Australia | >11,000 ^b | [1] |
| <i>Cordia curassavica</i> (Jacquin) Roemer and Schultes | Mauritius | 1,000 ^a | [9] |
| <i>Hypericum perforatum</i> L. | United States | >9,900 ^b | [5] |
| <i>Opuntia ficusindica</i> (L.) Miller | South Africa | 460 ^a | [10] |
| <i>Opuntia</i> spp. | Nevis, W.I. | 2,000 ^a | [9] |
| <i>Senecio jacobaeae</i> L. | United States | 1,400 ^b | [1] |
| <i>Salvinia molesta</i> DS Mitchell | Sri Lanka | >160,000 ^b | [1] |
| <i>Xanthium strumarium</i> L. | Australia | 130 ^a | [1] |

^aAnnual.

^bCumulative over the entire period of the active program.

of amino acids, the building blocks of proteins; 5) cell division inhibitors, which act during preemergence to disrupt mitosis in seedlings; 6) auxin mimics, which disrupt hormone balance to disrupt protein synthesis and cause a variety of growth abnormalities; 7) respiration inhibitors, which disrupt metabolism by uncoupling oxidative phosphorylation; and 8) herbicides of unknown mode of action, which include most inorganic, but also several organic, herbicides.

Herbicidal use can increase crop yields through improved weed control and by allowing earlier planting, both of which reduce the costs of production. Costs of herbicides in crop production are small relative to other energy inputs, such as fuel, nitrogen fertilizer, and irrigation. Herbicides require less energy than tillage for controlling weeds. Even in situations in which labor is plentiful and cheap, herbicides can be used profitably to control weeds in crop rows where mechanical methods are ineffective, and to provide early-season weed control when competition would result in the greatest crop yield reduction and when conditions, such as wet soil, would make other methods (e.g., cultivation) less effective or impossible to use. Herbicides reduce or eliminate the need for cultivation, which can injure crop roots and foliage, and can reduce the destruction of soil structure by reducing the need for tillage and by lessening the exposure of the soil to heavy machinery.

Herbicides can save labor and energy by reducing the need for hand weeding and mechanical tillage. By eliminating weed competition, they can reduce fertilizer and water use in crops. They can reduce harvest costs by eliminating interfering weeds, and can decrease grain drying costs through the elimination of green, weedy materials. Although other methods of weed control accomplish these things, they do not do so as efficiently or, often, as cheaply. Over the years, the cost of herbicides, relative to crop prices and labor and machinery costs, has decreased steadily; herbicides represent a control technology that is cheap, reliably effective, and provides consistent returns on investment.^[6]

Herbicidal use is not without disadvantages. Herbicides have varying acute mammalian toxicities. They may persist in the environment and contaminate food and groundwater, posing risks to human health.^[7] The use of selective herbicides may lead to the creation of secondary pests by eliminating some weeds, thus creating vacant niches into which other weeds may move. Weeds also may develop resistance to herbicides.

CONCLUSION

Weeds are a ubiquitous presence in the environment. Whereas losses caused by insects or plant pathogens

may vary in severity from year to year, the impact of weeds tends to be fairly constant. The homeowner, farmer, or rancher need not doubt that there will be weed problems every year. Weeds cause huge economic losses worldwide and require enormous inputs of labor, materials, and energy to combat.

Although various weed control technologies have been used for millennia, the concept of weed management, which is based on a more diverse approach to solving weed problems, is a much more recent development. Weed management takes into account the entire agricultural system, including climatic, edaphic, and biotic influences, and emphasizes optimal land use and maximum sustainable crop yield. A good weed management program should adopt a systematic approach to minimize weed impacts, combining prevention with control and employing techniques that are economically and environmentally sound. It should incorporate ecological principles; include economic thresholds; stress the use of cultural methods, such as plant interference and crop-weed competition, and, where appropriate, biological controls; and integrate several techniques, including the use of selective herbicides, into a cohesive control strategy.

REFERENCES

1. McFadyen, R.E.C. Biological control of weeds. *Annu. Rev. Entomol.* **1998**, *43*, 369–393.
2. Goeden, R.D. A capsule history of biological control of weeds. *Biocontrol News Inf.* **1988**, *9* (2), 55–61.
3. Julien, M.H., Griffiths, M.W., Eds.; *Biological Control of Weeds: A World Catalogue of Agents and Their Target Weeds*, 4th Ed.; CABI Publishing: Wallingford, UK, 1998.
4. Zimdahl, R.L. *Fundamentals of Weed Science*, 2nd Ed.; Academic Press: San Diego, CA, 1999.
5. Batra, S.W.T. Biological control of weeds: principles and prospects. In *Biological Control in Crop Production*; BARC Symposium 5; Papavizas, G.C., Ed.; Allanheld, Osmun: London, 1981; 45–59.
6. Bridges, D.C. Impact of weeds on human endeavors. *Weed Technol.* **1994**, *8* (2), 392–395.
7. Culliney, T.W.; Pimentel, D.; Pimentel, M.H. Pesticides and natural toxicants in foods. *Agric. Ecosyst. Environ.* **1992**, *41* (3/4), 297–320.
8. Andres, L.A. The economics of biological control of weeds. *Aquat. Bot.* **1977**, *3* (2), 111–123.
9. Simmonds, F.J. The economics of biological control. *J. R. Soc. Arts* **1967**, *115* (5135), 880–898.
10. Huffaker, C.B.; Simmonds, F.J.; Laing, J.E. The theoretical and empirical basis of biological control. In *Theory and Practice of Biological Control*; Huffaker, C.B., Messenger, P.S., Eds.; Academic Press: New York, 1976; 41–78.

Weed Management: Home Landscaping

Stephen C. Weller

*Department of Horticulture and Landscape Architecture, Purdue University,
West Lafayette, Indiana, U.S.A.*

INTRODUCTION

Weeds in the home landscape can cause many problems including reducing the esthetics of the planting and interfering with growth of the desirable landscape plants by competing for water, nutrients and sometimes light. Some weeds cause severe allergies (e.g., ragweed, poison ivy, poison oak) and can be a severe hazard to humans.

What exactly are weeds? Generally speaking, weeds are plants that are not desirable in a particular situation and, as such, need to be removed. Typically, however, the term weed refers to naturally occurring aggressive plants that are injurious to people or to agriculture. Weeds come in many different forms including summer and winter annuals, biennials, and perennials and are usually separated into grasses, sedges, and broadleaf plants. Summer annual weeds germinate in spring, grow and flower over summer, produce seed, and die in the fall when temperatures decrease. Winter annuals emerge in late summer and most overwinter as a rosette and after receiving a cold treatment, send up a seed stalk (bolt), flower, and produce seeds early in the summer. The exceptions are henbit and chickweed, which do not form rosettes, but overwinter as immature and mature plants. Biennials (such as wild carrot) live for 2 years and are similar in many ways to winter annuals except that they flower and produce seed only in the second year. Perennials grow for a number of years, sometimes indefinitely, and can be herbaceous (soft stemmed) or woody and reproduce by seed or vegetative (asexual) reproduction. Herbaceous perennials include field bindweed, yellow nutsedge, and quackgrass; woody perennials include trees, woody shrubs, and vines.

Weed management in the home landscape usually involves lawn care and care of shrubs, trees, and flowers. The essence of weed management in the home landscape involves prevention. This involves reducing the area where weeds can grow. Tactics used will change slightly depending on whether the ornamentals in the area are perennial (herbaceous or woody) or annuals. Weed management includes several steps: site assessment, site preparation, and implementation of weed management practices compatible with the species planted and the objectives of the homeowner.

Site assessment involves determining what plants will grow best in the area and what steps must be taken to adequately eliminate existing weeds to allow the successful performance of the subsequently planted ornamentals. Site preparation involves removal of undesirable weedy vegetation prior to planting of the landscape plants. Perennial weed elimination is critical in site preparation. When perennials are a major problem, the best method of elimination is the use of a non-selective systemic postemergence herbicide (applied to foliage of emerged weeds) such as glyphosate when weeds are actively growing. If no perennials exist at the site, the best approach is to properly prepare the soil for planting by addition of soil amendments that allow good tilth and fertility. Healthy, vigorously growing plants are better able to compete with weed invasions. The best weed control for lawns is to maintain a healthy vigorous turf. Weed management options in ornamentals, whether the plantings are trees or shrubs, woody groundcovers, annual flowers, herbaceous perennials, or mixed plantings of woody and herbaceous plants, include prevention and sanitation, mulches, geotextiles, handweeding, and in some cases, herbicides.

WEED MANAGEMENT OPTIONS FOR LAWNS

Yard grass weed management involves techniques that produce a healthy actively growing turf. Good management involves proper grass species selection for the region, then maintaining fertility, watering, and soil pH while removing accumulated thatch, and carrying out proper mowing frequency (every 7 days) and mowing height (2–4 in.). If a lawn is healthy and properly maintained, weeds can often be managed by hand pulling or cutting out. A weedy lawn is symptomatic of improper management, but when weeds are present, herbicides are often required.

There are many types of lawn herbicides to control broadleaf and grassy weeds. The most user-friendly and safe herbicides for the average homeowner are combined with granular fertilizer formulations and applied to wet foliage either in spring or in the fall. These weed and feed types can be purchased at most

garden centers for application with fertilizer spreaders that are easily calibrated for effective weed management. There are also many formulated liquid herbicides for postemergence broadleaf weed control in turf that are sold in hand spray bottles for spot treatment of problem weeds such as dandelions. Annual grassy weeds like crabgrass can be managed by applying a preemergence herbicide (a herbicide applied to the soil prior to weed seed germination and plant emergence) in late winter. Preemergence herbicides can interfere with establishing desirable turfgrasses from seed.

WEED MANAGEMENT OPTIONS FOR ORNAMENTALS

Home ornamental gardeners should concentrate on non-herbicidal approaches where possible for weed management. Although herbicides are effective in controlling a number of weeds in the home landscape, they must be carefully used and require precise application for optimal performance and to reduce the potential for causing injury to valuable ornamentals. Because of the wide variety of plants used in landscaping, it is seldom possible to use a single herbicide for all weed management since ornamentals vary significantly in their injury susceptibility to herbicides. Non-chemical methods when used properly are quite effective in managing weeds.

Prevention and sanitation will reduce future weed problems in the home landscape. Prevention involves not allowing the introduction of new weeds to the landscape. Installing weed-contaminated planting stock is a major source of weed problems for many homeowners. Therefore, purchase only annuals and perennials that are weed free. Any seeds produced by existing weeds will add to next season's weed problems. Sanitation involves removing all weedy plants from the site. Some annuals such as purslane can vegetatively propagate if left on the soil surface, where they may grow again.

Weeds are most problematic in the initial years of any ornamental planting, when plants are small. In established groundcover beds or dense planting of herbaceous perennials, weed problems decrease as the plants grow and cover the soil surface. In hedges or mixed plantings, weeds can be a persistent problem in the area not occupied by specimens.

Many homeowners use some type of organic mulch around ornamentals for weed control since they are effective and add an appealing esthetic appearance to the planting. Mulches have utility in all types of ornamental plantings. Organic mulch options include aged barks, various hulls, municipal composts, crushed

rocks, sawdust, leaves, and grass clippings. Bark mulch of aged softwood or hardwood species or cypress are the most commonly used mulches in ornamentals since they are coarse-textured, have a low water-holding capacity, and do not decompose or settle rapidly. These mulches are applied 3–4 in. thick on the soil surface and last throughout the entire season and prevent weeds from germinating due to their ability to reduce light on the soil surface. Supplemental weeding by hand or with a carefully applied spot spray of a non-selective herbicide (one that kills all plants) is sometimes necessary to remove some weeds. Compost, sawdust, leaves, and grass clippings are not as attractive, decompose rapidly, and are not as effective against weeds as bark mulches. Grass clippings taken from herbicide-treated lawns can be toxic to the ornamentals. Use of crushed rocks is not as common in the home landscape but if used should be laid on a solid sheet of polyethylene mulch to prevent weed germination and growth; otherwise, weeds become a major problem. However, plastic mulch may cause problems with water availability for ornamentals plantings.

Geotextiles are synthetic fabrics that cover the soil surface but allow movement of water and air while reducing the light reaching the soil surface, which reduces weed germination and growth. Although these materials are expensive and require installation, they become more cost-effective over time since they last 4 years or longer. They are as effective as a preemergence herbicide which requires reapplication each year. Geotextiles are used on perennial plantings that do not require yearly replanting but are unsuitable for spreading groundcover beds since the fabric inhibits plant rooting. Geotextiles must be covered by a mulch to reduce photodegradation and improve the appearance of the beds. Any weeds growing through the textiles should be quickly removed to prevent holes in the fabric barrier.

Hand weeding is a commonly used non-chemical method of weed management. If used as the sole tool for weed management, hand weeding is laborious and excessively time consuming. Most avid gardeners use hand weeding as a supplement to mulching and use of landscape fabrics.

Although the herbicide option is always available for the home landscape, it should not be the only tool used. If herbicides are used by the homeowner, they should be combined with non-chemical methods and use should be based on advice from professional pest managers. There are many sources of information regarding herbicide use in ornamentals available through state extension services. The homeowner should strive to become informed on which herbicides are available for use, which plants the herbicides can be used on, which weeds the herbicides do or do not control, and how the herbicides must be applied to work and be safe

in the environment. Many homeowners can use appropriate postemergence herbicides as spot sprays on weeds found in sidewalks, in driveways, or as isolated patches in ornamental plantings. Non-selective, postemergence herbicides, such as glyphosate or pelargonic acid, are safe and easy to use, although application to foliage of ornamentals must be avoided as these chemicals will cause injury. In all cases where herbicides are used, it is important to read the entire product label and follow all instructions on mixing, application, and disposal for effective and safe use.

BIBLIOGRAPHY

- Bloetscher, B.; Boehm, M.J.; Rimelspach, J.W.; Shetlar, D.J.; Street, J.B. *Management of Turfgrass Pests: Weeds, Diseases and Insects*; Bulletin L-187-01; Cooperative Extension Service, The Ohio State University: Wooster, OH, 2001.
- Hart, S.E.; Lycan, D.W.; Meade, J.A. *Weed Control Around Home Grounds*; Fact Sheet 020; Rutgers Cooperative Extension, Rutgers University: New Brunswick, NJ, 2001.
- Monaco, T.J.; Weller, S.C.; Ashton, F.M. *Weed Science. Principles and Practices*, 4th Ed.; John Wiley and Sons, Inc.: Hoboken, NJ, 2002.

Weed Management: Introduction and Mechanical and Cultural Approaches

Thomas W. Culliney

Center for Plant Health Science and Technology, USDA, APHIS, PPQ,
Raleigh, North Carolina, U.S.A.

INTRODUCTION

A weed is a plant growing where it is not wanted. This is a purely anthropocentric view, of course, based on human aesthetics, economics, or health concerns; although the origin of plants predates considerably that of humans, no weeds existed on the Earth before the arrival of man. Worldwide, about 200 plant species are sufficiently troublesome to be classified as weeds.^[1] The most consistent trait shared by weedy species is the ability to colonize and thrive in habitats disturbed by human activity. Characteristics common to weeds almost inevitably bring them into conflict with human interests (Table 1). However, a weed under one set of circumstances may be an economically or ecologically valuable plant under another. Some positive qualities of weeds include: protecting otherwise bare soil from erosion; adding organic matter to the soil; providing food and cover for wildlife; yielding useful drugs, fuels, or other chemicals; serving as food in the human diet or forage for livestock; and beautifying the landscape. Some species now considered weeds, such as *Avena fatua* L. (wild oat), *Chenopodium album* L. (common lambsquarters), and *Camelina sativa* Crantz (false flax), were once cultivated as crops.^[2]

NEGATIVE IMPACTS OF WEEDS

Weeds impose economic, social, and medical costs in a number of ways. They compete with valued plants (food and fiber crops, ornamentals, and timber) for nutrients and sunlight, and additionally reduce crop yields through allelopathy (the release of chemical growth inhibitors into the soil); increase crop production costs through increases in mechanical and chemical controls; increase crop protection costs by harboring other pests (arthropods, nematodes, and plant pathogens); reduce the quality of farm products (e.g., through contamination of crop seed with weed seed, and reduction in the area of useful forage for livestock in pastures and rangelands); reduce the quality of livestock or game through toxicity, leading to death or failure to thrive, negative effects on the quality of animal products (milk, fleece, or hides), or reproductive

failure; increase processing costs incurred in cleaning contaminated products; interfere with water management in agriculture by infesting irrigation ditches; increase costs to maintain rights-of-way for railroads, highways, and power and telephone transmission lines; endanger human health and cause loss of labor productivity by causing allergies and poisonings; and decrease land values. Economic losses because of weeds exceed those caused by any other class of agricultural pest. The direct costs to agriculture from crop losses and the imposition of control measures have been estimated in the range of US\$15–32 billion annually in the United States alone.^[3,4] Where weeds are not controlled, crop losses may reach as high as 90%.^[5]

WEED MANAGEMENT

Weed management combines the techniques of prevention, eradication, and control to eliminate or mitigate weed problems in a crop, cropping system, or wider environment.^[6] Factors such as a field's cropping history, a grower's management objectives, and available technology and financial resources all are taken into account to support good management decisions. The term *weed management* suggests a shift away from a strict reliance on control of existing weed problems to greater emphasis on the prevention of propagule (seed, spore, etc.) production, reduction of weed emergence in the crop, and minimization of weed competition with the crop. Weed management emphasizes an integration of techniques and knowledge of weed biology in a way that considers the causes of weed problems, rather than provides a mere reaction to them once they arise.^[7] As various environmental and cultural factors affect the weed–crop balance, the ultimate aim of weed management is to tip the balance in favor of the crop by manipulating these key factors.

A good weed management program emphasizes prevention over control. The first rule of weed prevention is the use of clean seeds. Other preventive practices may include:^[6] 1) isolation of newly introduced livestock to prevent spread of weed seeds caught in feathers or hair, or from their digestive tracts; 2) use of clean farm equipment and cleaning of itinerant equipment;

Table 1 Common characteristics of weeds (Not all weeds exhibit all characteristics)

| |
|---|
| Rapid seedling growth and early reproduction |
| Rapid maturation or a brief time spent in the vegetative stage |
| Potential for dual modes of reproduction—seed production and vegetative reproduction |
| Environmental plasticity—capable of tolerating and growing under a wide range of climatic and edaphic conditions |
| Self-fertility, although selfing is not obligatory |
| Pollination accomplished by non-specific flower visitors or by the wind, if cross-pollinated |
| Resistance to adverse environmental conditions (e.g., having seeds that remain dormant in the soil and resist decay for long periods) |
| Germination requiring no special environmental conditions |
| Production of seed of the same size and shape as crop seed, thus facilitating spread by man |
| Ability to produce large numbers of seed and produce at least some seeds over a broad range of environmental conditions |
| Possession of specially adapted seed dispersal mechanisms |
| Ability of seedlings to root and emerge from deep in the soil |
| Possession of roots and other vegetative organs (in perennials) capable of vigorous growth, with large food reserves, promoting survival under adverse environmental conditions and intensive cultivation |
| Rapid regeneration of severed vegetative organs |
| Adaptations to withstand or repel grazing |
| Competitive ability enhanced by special attributes (e.g., rosette formation, climbing growth, and allelopathy) |
| Ability to evolve resistance to control measures |

Source: (From Refs.^[5,6])

3) cleaning of irrigation water; 4) maintenance of sanitation on irrigation ditch banks; 5) inspection of imported nursery stock for presence of weeds, their seeds, and vegetative reproductive organs; 6) inspection and cleaning of imported gravel, sand, and soil; 7) surveillance of fence lines, field edges, rights-of-way, and railroads as potential sources of new weeds; 8) prevention of the deterioration of range and pasture lands to preclude the easy entry and establishment of weeds; and 9) guarantee that seed dealers and grain handlers clean crop seeds and dispose of the contaminants properly.

If prevention fails, control measures must be utilized to minimize the economic impact of a weed infestation. Although various weed control methods have been developed and used over thousands of years, none has ever been abandoned completely; new techniques have been added through technological innovation,

but old ones still are used effectively, especially in small-scale agriculture.^[6] There are four principal categories of weed control: mechanical, cultural, biological, and chemical.

Mechanical Control

Mechanical methods encompass two major modes of action: disturbing the soil to bury seeds or loosen or cut the root system, and cutting above-ground parts of the weed. Mechanical controls include: 1) hand pulling—effective for annual weeds (establish and reproduce within a single growing season), but not for perennials (weeds that live for 3 years or more) capable of vegetative reproduction; 2) hand hoeing—controls most persistent perennial weeds if done regularly; most effective where labor is abundant and cheap; 3) tillage—may be the most economical method; controls weeds by burying them, separating shoots from roots, stimulating germination of dormant seeds and vegetative buds (to be destroyed by subsequent tillage), desiccating shoots, and exhausting the carbohydrate reserves of perennial weeds. The success of tillage depends on various biological factors. Weeds that share a crop's growth habit and time of emergence, or that germinate over a long time period, may be most difficult to control, as are perennial weeds that reproduce vegetatively; 4) mowing—by removing shoot growth, it prevents seed set and may deplete root reserves in some upright perennials, but is ineffective against prostrate types; and 5) draining—effective in controlling weeds, such as cattails and bulrushes, that grow best in wet environments.

Cultural Control

Manipulation of the crop environment in space and time can create conditions that favor growth of the crop over that of weeds. These include: 1) crop competition—varying the cropping pattern and intercropping. Increasing crop density and biomass through increases in seeding rates or decreasing row spacing, enabling the crop to form a closed canopy providing heavy shade, can suppress weed growth. Intercropping (polyculture), the simultaneous culture of two or more crops on the same plot of land, can be used for the competitive suppression of weeds.^[8] Increasing the complexity of the cropping system by interplanting crops of different growth form, phenology, and physiology creates a pattern of resource use different from that in monoculture, which can lead to the preemptive use of resources by the intercrop at the expense of the weed. Allelopathy also may play a role in weed suppression by intercrops. Intercropping seems particularly effective in controlling weeds under conditions

of low soil fertility, in which intercrops acquire a greater share of available nutrients; 2) planting date—early planting can provide crops a competitive edge over weeds because of their earlier establishment; 3) companion cropping—cover crops or living mulches may be used as companion plants to suppress weeds in crop fields. They have the added benefit of adding nutrients to the soil (e.g., where legumes are used), improving water percolation, and reducing soil erosion; 4) crop rotation—as some weed species are associated with certain crops more than with others, merely changing the crop can alleviate some weed problems. Control may be achieved by crop–weed competition, or in conjunction with the different cultural methods employed with the new crop.

REFERENCES

1. Holm, L.; Doll, J.; Holm, E.; Pancho, J.; Herberger, J. *World Weeds: Natural Histories and Distribution*; John Wiley and Sons: New York, 1997.
2. Altieri, M.A. The impact, uses, and ecological role of weeds in agroecosystems. In *Weed Management in Agroecosystems: Ecological Approaches*; Altieri, M.A., Liebman, M., Eds.; CRC Press: Boca Raton, FL, 1988; 1–6.
3. Bridges, D.C. Impact of weeds on human endeavors. *Weed Technol.* **1994**, *8* (2), 392–395.
4. Pimentel, D.; Lach, L.; Zuniga, R.; Morrison, D. Environmental and economic costs of nonindigenous species in the United States. *Bioscience* **2000**, *50* (1), 53–65.
5. Batra, S.W.T. Biological control of weeds: principles and prospects. In *Biological Control in Crop Production*; BARC Symposium 5; Papavizas, G.C., Ed.; Allanheld, Osmun: London, 1981; 45–59.
6. Zimdahl, R.L. *Fundamentals of Weed Science*, 2nd Ed.; Academic Press: San Diego, CA, 1999.
7. Buhler, D.D.; Liebman, M.; Obrycki, J.J. Theoretical and practical challenges to an IPM approach to weed management. *Weed Sci.* **2000**, *48* (3), 274–280.
8. Liebman, M. Ecological suppression of weeds in intercropping systems: a review. In *Weed Management in Agroecosystems: Ecological Approaches*; Altieri, M.A., Liebman, M., Eds.; CRC Press: Boca Raton, FL, 1988; 197–212.

Weed Management: Ornamental Nurseries

Stephen C. Weller

*Department of Horticulture & Landscape Architecture, Purdue University,
West Lafayette, Indiana, U.S.A.*

INTRODUCTION

Weeds are a major management consideration for all landscape ornamental nursery operators. Crops grown in nurseries include field- and container-grown ornamentals. Weeds compete with the ornamental crops for all essential growth factors (light, water, nutrients, space) and if left uncontrolled, result in weak and unmarketable ornamental plants. Ornamentals containing weeds are unattractive and will inhibit sales, and when sold to the consumer, contaminate the home landscape, creating problems for the homeowner. Consequently, it is imperative for nursery operators to have effective ornamental weed management programs in place.

A balanced nursery field and container crop weed management program involves using an integration of tools that maintains the site as weed-free as possible. A long-term approach for most sites often involves a 2–3 yr program from the time the ornamentals are planted until they are sold to the consumer. In a well-managed nursery, the most troublesome weeds are annuals that complete their lifecycle in 1 yr, but they are the most easily controlled. Perennial weeds are the most difficult to control and manage and can create greater problems in the overall weed management program.

METHODS OF WEED MANAGEMENT IN NURSERIES

The components of an integrated weed management approach for field nurseries involve prevention and sanitation, hand weeding and cultivation, mulches, and herbicides. Weed management in container nurseries involves integration of these techniques, but there is less flexibility.

Prevention and sanitation involves several approaches to reduce initial and longer-term weed problems. In field nurseries, the first step of prevention is the elimination of all previously existing

perennial weeds. Nurseries that start with a weed-free site are the most successful, and such an approach leads to easier weed management throughout the life of a plant. Prevention can involve fumigation with chemicals such as metham that eliminate most weeds and seeds. Other prevention approaches include prior site management with an agronomic crop (corn or soybean) plus herbicides, fallowing and treatment with non-selective herbicides, or cover cropping (small grains or green manure crops) to smother weeds. In container nurseries, growers should use soils free of weeds or seeds, and potting mixes that are certified weed-free. Container nursery operators often use black polyethylene mulch on the soil surface to eliminate soil weeds emerging between containers. Forms of sanitation for field and container crops include planting weed-free stock plants, cleaning nursery equipment after each use, and reducing seed production by existing weeds through mowing, hand pulling-rouging, or herbicide sprays. All weeds capable of vegetative reproduction must be removed from the site after hoeing or pulling.

Hand pulling and cultivation involves removing all weeds by use of manual or machine labor. Hand pulling is often necessary in containers if emerged weeds are present even though it is tedious and time consuming since there are no selective herbicides labeled for use in containers that control emerged broadleaf weeds. Cultivation is used in field production, although care has to be taken not to damage crop roots or stems when using rototillers, disks, plows, and hoes. Several types of specialized field cultivation equipment are available that reduce or eliminate trunk damage and allow within-row weed cultivation (finger and torsion weeders). Mowing is often used in field production to reduce weed seed production or growth of existing weed populations.

Cover crops are sometimes used during the field nursery cycle to reduce the weed presence. Crops such as small grains (oats, annual rye, wheat) or legumes can be planted within and between rows and maintained as a living mulch by mowing or killed by a herbicide and left on the soil surface as an organic mulch to inhibit weed seed germination and growth.

Table 1 Common names of herbicides labeled for ornamental use^a

| Preplant to all ornamentals | Preemergence | Postemergence |
|-----------------------------|-----------------------------|--|
| Methyl bromide | Benefin + oryzalin | Asulam |
| Dazomet | Bensulide | Bentazon |
| Diquat | DCPA | Clethodim |
| Glufosinate | Diclobenil | Clopyralid |
| Glyphosate | Dithiopyr | Diclobenil |
| Paraquat | Imazaquin | EPTC |
| Pelargonic acid | Isoxaben | Fenoxaprop |
| | Isaxaben + trifluralin | Fluazifop |
| | Metolachlor | Glyphosate (no green bark or foliage) |
| | Napropamide | Halosulfuron |
| | Napropamide + oxadiazon | Imazaquin |
| | Norflurazon | Oxyfluorfen |
| | Oryzalin | Paraquat |
| | Oxadiazon | Sethoxydim |
| | Oxyfluorfen | |
| | Oxyfluorfen + oryzalin | |
| | Oxyfluorfen + oxadiazon | |
| | Oxyfluorfen + pendimethalin | |
| | Prodiamine | |
| | Pronamide | |
| | Prodiamine + oxadiazon | |
| | Simazine + pendimethalin | |
| | Trifluralin | |

^aSee label for specific ornamentals and uses. Always read and follow all instructions on the herbicide labels.

Mulching with natural inorganic or organic materials or synthetic mulches is not common in commercial field nurseries, although quite common in the home landscape and in parks or other public areas. The exception is in container areas where synthetic mulches are common under the containers. The synthetic mulches used include polyethylene mulches (usually black) or fabric mulches (weed mats). The main purpose of mulch in container areas is to prevent weeds from germinating and becoming a problem between containers.

Herbicides are the main weed management tool of choice in field and container ornamental nurseries. Herbicides used include preemergence (applied to the site prior to weed emergence) and postemergence (applied to weed leaf foliage after emergence) (Table 1). The use and choice of a preemergence herbicide involves knowledge of the weeds at the site, the types of ornamentals grown, and the effectiveness and selectivity of the herbicide choices available for use. Selectivity of many herbicides for ornamentals is achieved by formulation. Formulation of the herbicide as a granule is the best choice since this allows application without

contact of the herbicide on the crop foliage, which reduces injury potential. When preemergence herbicides are used, operators should make sure that all emerged weeds are removed from the site prior to application. After application, water should be applied to the soil to move the herbicide into the weed seed germination zone. Application of most preemergence herbicides after weed seed germination and emergence results in lowered or no weed control. Many of the pre-emergence herbicides available for field nurseries will provide acceptable weed control for most of the growing season but most are only effective for 8–12 weeks in containers. In containers, emerged weeds are generally hand-pulled from the pots prior to reapplication of herbicides.

Postemergence herbicides are available for most weeds in field-grown ornamentals, but only selective grass herbicides are available for use in containers; no selective postemergence broadleaf weed herbicides are labeled for containers. In field nurseries, there are several types of selective and non-selective postemergence herbicide options (Table 1). In many instances, the most commonly used herbicides are the

non-selective types that are applied as directed sprays to the weeds or as spot sprays on weedy patches. Herbicides such as glyphosate, glufosinate, paraquat, or pelargonic acid are the most commonly used to kill emerged annual and perennial weeds. Growers must use extra caution when applying such herbicides since they can injure the ornamentals; thus, spray applications are either directed at the base of the woody ornamentals or applied with shields.

CONCLUSIONS

Weed control in nurseries is based on an integrated approach using a variety of methods. The key is to start with a weed-free site and maintain the site as weed-free as possible. Once weeds become established,

the job of management becomes difficult and expensive. For additional information on weed management in nurseries contact local county extension offices, local nursery management companies, or access university extension weed management guides available on the Internet.

BIBLIOGRAPHY

- Monaco, T.J.; Weller, S.C.; Ashton, F.M. *Weed Science. Principles and Practices*, 4th Ed.; John Wiley and Sons, Inc.: Hoboken, 2002.
- Kuhns, L.J.; Harpster, T.; Rose, M.A.; Guiser, S. *Controlling Weeds in Nursery and Landscape Plantings*; Publications Distribution Center, The Pennsylvania State University: University Park, PA, 1997.

Weed Seed Dormancy: Implications for Weed Management Strategies

Diego Batlla

Departamento de Produccion Vegetal, Universidad de Buenos Aires, Buenos Aires, Argentina

Roberto Benech-Arnold

C.O.N.I.C.E.T./Facultad de Agronomía, Universidad de Buenos Aires, Buenos Aires, Argentina

INTRODUCTION

Dormancy is a common attribute of many weed species and is probably the most important of a series of processes that determine the seasonal annual pattern of weed emergence commonly observed under field conditions.^[1] A new general definition of dormancy was recently proposed: "Dormancy is an internal condition of the seed that impedes its germination under otherwise adequate hydric, thermal and gaseous conditions."^[2] This implies that, once dormancy has been removed, seed germination would proceed under a wide range of environmental conditions.

Under field conditions, weed seed dormancy is regulated by a complex interaction of environmental factors (i.e., temperature, soil water status, light, etc.). Most agronomic practices can affect these factors by altering the physical environment to which weed seed banks are exposed. Thus knowledge about the ways in which environmental factors and agronomic practices affect the dormancy status of weed seeds could be used to develop and improve weed management strategies.

CONCEPTUALIZING WEED SEED DORMANCY

Dormancy is classified into primary and secondary dormancy. Primary dormancy refers to the innate dormancy present in the seeds when they are dispersed from the mother plant. Secondary dormancy refers to a dormant state that is induced in non-dormant seeds, or reinduced in once-dormant seeds after a sufficiently low dormancy had been attained, by unfavorable conditions for germination. The release from primary dormancy followed by subsequent entrance into secondary dormancy may lead to dormancy cycling. Evidence for dormancy cycling has been obtained for seeds of many weed species under field conditions.^[2,3] Generally, seeds are released from dormancy during the season preceding the period of favorable conditions for

seedling and plant development, whereas dormancy is induced in the season preceding the period that is harmful for plant survival. For example, several summer annual species exhibit a high dormancy level in autumn; during winter they undergo dormancy relief, but dormancy increases again during summer (Fig. 1). On the contrary, winter annual species generally show the reverse dormancy pattern. Therefore the patterns of dormancy are of high survival value to weed species determining germination under environmental conditions that will ensure growth and reproduction. Normally, seedling emergence in the field occurs when the dormancy level of the seed population is at its minimum (Fig. 1).

Changes in dormancy status of weed seed populations are associated with changes in the range of temperatures and water potentials permissive for seed germination. As dormancy is released, the range of temperatures and water potentials permissive for germination widens until it is maximal; on the contrary, as dormancy is induced, the range of temperatures and water potentials over which germination can proceed narrows, until germination is no longer possible at any temperature or water potential. Germination in the field is therefore restricted to the period when the field temperature and soil water potential, and the temperature and water potential range over which germination can proceed, overlap.

However, in many weed species once environmental temperature and water potential are within the permissive range, dormancy must be terminated by the effect of additional environmental factors for the germination process to proceed. In these cases, changes in the degree of dormancy not only comprise changes in temperature and water potential requirements for germination, but also in sensitivity of the seed population to the effect of those dormancy-terminating factors. Fluctuating temperatures and light are two critical environmental factors that can trigger dormancy termination in seeds of many weed species. An ecological interpretation of this requirement to

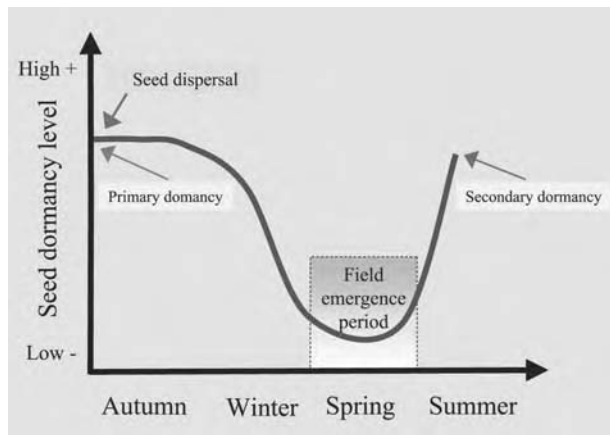


Fig. 1 Schematic representation of seasonal changes in the seed population dormancy level for a summer annual weed.

complete exit from dormancy in certain weed species has been related to the possibility of detecting canopy gaps as well as depth of burial under field situations.^[2]

Dormancy cycles observed in some species are known to be regulated mainly by temperature in temperate environments where water is not seasonally restricted. For example, in summer annual species dormancy relief is produced by the low temperatures experienced during winter, whereas high temperatures enhance their dormancy level during summer. Several winter annual species show the reverse dormancy pattern. Hence high temperatures during summer result in dormancy relief, and low temperatures during winter can induce secondary dormancy. Although much experimental data support the main role of soil temperature as regulator of seed dormancy, there is evidence indicating that the effect of temperature on dormancy release and induction may be modulated by soil moisture conditions.^[2]

Although the situation under actual field conditions is far more complicated because of the many

environmental factors that affect seed dormancy, the presentation of material so far is a useful simplification for understanding how the environment controls dormancy in weed seed banks. Fig. 2 illustrates the conceptual framework derived from the above definitions of the different factors affecting dormancy in weed seed populations. The chart aims to illustrate the different “pathways” that a seed population could undergo; hence passage along the whole flowchart is by no means the only possibility. For example, a seed population might be dispersed with a low level of dormancy and might or might not require limited stimuli for dormancy termination. In this case, the population would not experience the left-hand side of the flowchart and would or would not bypass the “zone” of dormancy termination.

USING WEED SEED DORMANCY CHARACTERISTICS TO DESIGN WEED CONTROL STRATEGIES

Knowledge about seed dormancy characteristics of weed populations could help in identifying agronomic practices that result in very low seedling establishment, even when a high density of weed seeds is present in the field.^[4] Many weed seeds require light to terminate dormancy and give way to the germination process; consequently, the light environment could be managed to impede seed germination. Light signals are perceived by seeds through the phytochrome system. Generally, low-red/far-red wavelength ratios inhibit seed germination, whereas high-red/far-red wavelength ratios promote seed germination. Light filtered by green leaves is rich in far-red wavelengths and explains the low-red/far-red wavelength ratios measured under plant canopies. Therefore plant cover could be managed to reduce some weed problems. For example, changing plant architecture, crop-sowing densities,

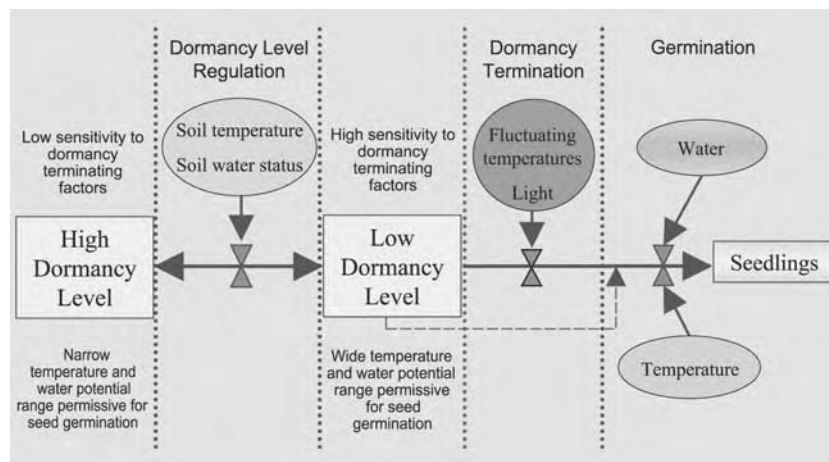


Fig. 2 Flowchart representing changes in dormancy level and termination of dormancy in seed populations and the environmental factors that most likely affect each process.

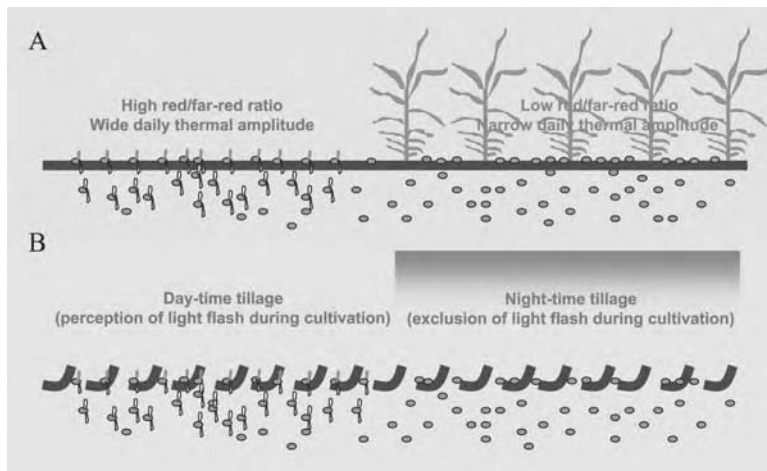


Fig. 3 Schematic representation of two different strategies for reducing weed seedling establishment in agricultural systems: (A) use of plant cover to reduce red/far-red wavelength ratio and the amplitude of the temperature fluctuation regime to which weed seed banks are exposed, and (B) performing tillage at night time to exclude seeds perception of light flash during cultivation.

crop-plant spacing, the use of cover crops, and intercrops may have a high potential for improving weed management by preventing the exit of weed seeds from dormancy and for germination to occur under field situations (Fig. 3A).

Another source of light for buried weed seeds is the brief light pulse received during cultivation. Many weed seeds buried in the soil acquire an extremely high light sensitivity that permits them to detect submilli-seconds of sunlight when the soil is disturbed. This is reflected in the high weed-emergence rates following tillage operations. This suggests that germination of light-requiring seeds would be impeded if a non-tillage crop-production system is implemented or if cultivation is performed at night.^[4,6] In some studies, a significant reduction in weed emergence was observed in plots cultivated at night in comparison to emergence levels obtained under daytime cultivation^[5,6] (Fig. 3B). Another possibility would be to add additional light sources during cultivation to stimulate weed emergence and thus help deplete weed seed banks.^[6]

The other environmental factor that usually terminates dormancy in many weed seeds under field conditions is temperature fluctuation. Generally, seed germination responses are positively related to temperature fluctuation amplitude, large temperature fluctuation regimes determining dormancy breakage of a higher proportion of the population than that observed under small temperature fluctuation regimes. Thus changing the amplitude of temperature fluctuation regimes in field environments would lead to a reduction in weed emergence. Plant cover or crop residues could be managed to achieve this objective, reducing the daily thermal amplitude to which weed seed banks are exposed (Fig. 3A). In the soil profile, temperature amplitude decreases with depth, so another possibility is to manage tillage to bury the seeds at a depth where temperature fluctuations are reduced enough to impede dormancy breakage.^[4]

The construction of weed emergence models that predict which proportion of the seed bank emerges at a given time would be a useful tool for determining the most suitable time for seedling control and, consequently, should result in a higher efficacy of controls methods. As pointed out earlier, changes in weed seed bank dormancy level are probably the most important process determining weed seedling emergence patterns in the field. Thus to predict time and proportion of weed seed bank emergence, we should consider changes in dormancy as affected by environmental factors in the construction of our germination models. For this purpose, we have to establish functional relationships between environmental factors regulating dormancy and dormancy changes of weed seed populations. For example, prediction of changes in light sensitivity of buried seeds would be a useful tool for determining tillage timing to control weed seed populations. Accurate predictions of changes in light sensitivity of buried weed seeds would permit better planning of tillage operations to determine the emergence of a high fraction of the seed bank population, which subsequently could be controlled by mechanical or chemical strategies; this should increase the efficacy of control methods and the corresponding impact on the seed bank population. Alternatively, tillage operations could be performed when the seed population has a low light sensitivity, diminishing the emergence of weeds from the seed bank prior to crop planting resulting in a reduction in herbicide applications.

CONCLUSION

With increased pressure to reduce pesticide inputs in agricultural systems, optimal timing and rates of chemical products as well as finding sustainable nonchemical alternatives for weed control will be paramount.^[7] In this chapter, we show that a better understanding of

how environmental factors and agronomic practices affect the dormancy status of weed seed banks could be used to develop and improve weed control strategies to meet this challenge.

REFERENCES

1. Forcella, F.; Benech-Arnold, R.L.; Sánchez, R.A.; Ghera, C.M. Modelling seedling emergence. *Field Crops Res.* **2000**, *67*, 123–139.
2. Benech-Arnold, R.L.; Sánchez, R.A.; Forcella, F.; Kruk, B.C.; Ghera, C.M. Environmental control of dormancy in weed seed banks in soil. *Field Crops Res.* **2000**, *67*, 105–122.
3. Baskin, C.C.; Baskin, J.M. *Seed Dormancy and Germination: Ecology, Biogeography and Evolution*; Academic Press: San Diego, CA, 1998.
4. Ghera, C.M.; Martinez Ghera, M.A.; Benech-Arnold, R.L. The use of seed dormancy to improve grain production. *J. Prod. Agric.* **1997**, *10*, 111–117.
5. Scopel, A.L.; Ballaré, C.L.; Radosevich, S.R. Photostimulation of seed germination during soil tillage. *New Phytol.* **1994**, *126*, 145–152.
6. Dyer, W.E. Exploiting weed seed dormancy and germination requirements through agronomic practices. *Weed Sci.* **1995**, *43*, 498–503.
7. Grundy, A.C. Predicting weed emergence: a review of approaches and future challenges. *Weed Res.* **2003**, *43*, 1–11.

Weeds and Carbon Dioxide

Lewis H. Ziska

Crop Systems and Global Change Laboratory, United States Department of Agriculture (USDA-ARS), Beltsville, Maryland, U.S.A.

INTRODUCTION

As global population continues to rise, demand for energy and food will increase concurrently. As a consequence, fossil fuel burning and deforestation will continue to be human-derived sources of atmospheric carbon dioxide. Since the 1950s, direct measurements of carbon dioxide concentration [CO_2] have shown an increase of approximately 20%, from 311 parts per million (ppm) to 380 ppm.^[1] The observed increase in [CO_2] ($\sim 0.5\%$ per year) is ongoing and global [CO_2] is expected to exceed 600 ppm by the end of the current century.^[2]

Although the association between rising [CO_2] and global warming has been emphasized, carbon dioxide is also the sole source of carbon for photosynthesis, and the continuing increase in atmospheric [CO_2] should result in a stimulation of plant growth. However, there are over 250,000 plant species, and it seems unlikely that rising carbon dioxide levels will stimulate photosynthesis and promote growth in exactly the same manner by species with no net effect on plant competition or success. For example, based on known biochemical subtypes, plants with C_3 photosynthesis (about 95% of all plant species, e.g., all trees) are more likely than plants with C_4 photosynthesis (about 4% of all plant species, e.g., corn) to respond to increasing [CO_2].^[3]

But is not more plant growth beneficial to human systems in any case? Critics of global warming point to the likely stimulation of plants by [CO_2] as a “wonderful and unexpected gift from the industrial revolution.”^[4] Yet, while there are obvious benefits to agronomically important crops, all plants are not equally desirable. What likely impacts can we anticipate regarding the growth and success of undesirable, or weedy species as [CO_2] increases?

WEED–CROP INTERACTIONS IN AGROECOSYSTEMS

Historically, weeds have always been associated with interference in crop production. Cultivation of agronomically desirable plants has led to inadvertent selection

for weedy species that mimic crop phenology. Early global classification of weeds by Holm et al.^[5] indicated that a majority of the world’s “worst” weeds had C_4 photosynthesis, whereas of the 86 crop species that make up 95% of the world’s food supply, only five are C_4 . Given what is known regarding the response of these different photosynthetic subtypes to rising [CO_2], this initially suggested that crop loss owing to weedy competition should decline in response to carbon dioxide.

Such an initial perspective now appears overly simplistic. Crop–weed competition varies by region; and C_3 and C_4 crops will interact with C_3 and C_4 weeds. Furthermore, a C_3 crop vs. a C_4 weed interpretation does not address weeds and crops with the same photosynthetic pathway; yet, many of the worst/troublesome weeds for a given crop are genetically similar with the same photosynthetic type [e.g., sorghum and johnson grass (C_4); oat and wild oat (C_3)]. Although data regarding interactive outcomes between crops and weeds are scarce, as [CO_2] increases, crops appear to be only favored where the weed is C_4 and the crop C_3 . All other data suggest a greater competitive ability on the part of the weed as [CO_2] increases (Fig. 1).

INVASIVE WEEDS

Clearly, agriculture necessitates an economic cost associated with weed–crop competition. However, human activities have also increased the number of new plant species introduced into unknown areas. While most are beneficial, a few result in widespread environmental damage and have been deemed invasive or noxious weeds. Clearly, it is crucial to recognize those factors that contribute to their biological success.

Is the rise in [CO_2] one such factor? A recent review^[7] suggested that on average, the growth response of individual invasive species to recent [CO_2] changes (those since the mid-19th century vs. current levels) and projected [CO_2] increases (current levels vs. the end of the 21st century) are about three and two times that of the published average, respectively, for all individual plants. While this suggests a “stronger than expected” response of individual invasive species, it

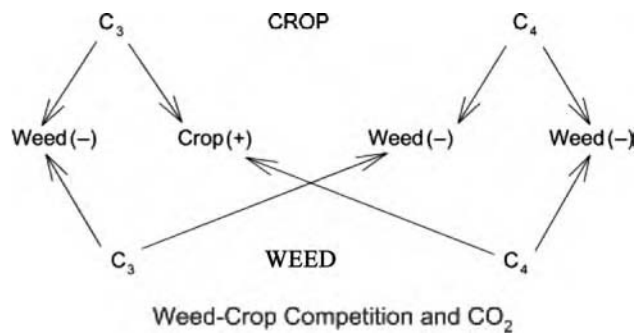


Fig. 1 Potential weed-crop interactions with increasing CO₂ above current levels as a function of photosynthetic subtype (either C₃ or C₄). Data are based on competition studies of crop and weed species and reflect either vegetative or reproductive increases. (From Ref.^[6])

is their aggregate response within a community that provides the best estimate of whether rising [CO₂] is increasing the success of weedy invaders. To date, four of five studies have indicated that [CO₂] can preferentially increase the growth of invasive plants within a community.^[7]

HUMAN HEALTH

The connection between weed biology and human health may seem esoteric to many, as plants are not disease vectors. However, there are a number of ways in which weeds impact human health, and it seems a fair question to ask how rising CO₂ could, potentially, alter these impacts.

Direct effects linking weeds and health include allergies, contact dermatitis, physical injury, and toxicity. Several studies have shown that rising CO₂ can stimulate ragweed pollen production.^[8] This has obvious implications with respect to allergic rhinitis, a disease that affects over 30 million people in the United States. Over 100 weedy species are also known to possess chemical irritants that induce dermatitis on contact. Among these are such well-known irritants as poison ivy and stinging nettle. At present, however, the impact of [CO₂] on either the growth or chemical content of these species is unknown. Many weeds also possess spines or sharp appendages that can puncture the skin. For example, Canada thistle, a noxious weed of North America, is noted for its leafy spines. At least one investigation has indicated that rising [CO₂] can alter the length and number of its spines, depending on leaf age.^[9] Lastly, there are over 700 plant species that are toxic to humans. Although the interaction between rising [CO₂] and the production of the specific poison is unclear, it is recognized that [CO₂] results in a stimulation of growth for many of these species.^[8]

IMPLICATIONS FOR WEED MANAGEMENT

Chemical Management

It has long been recognized that abiotic elements such as temperature, wind speed, soil moisture, humidity, etc. can alter chemical management; however, there are an increasing number of studies that demonstrate a decline in chemical efficacy with rising [CO₂] per se (Fig. 2). The basis for the decline in efficacy is unclear. Theoretically, because rising [CO₂] reduces stomatal aperture and/or number, it could reduce foliar absorption of herbicides. Timing of application could also be affected if increasing [CO₂] decreases the time the weed spends in the seedling stage (i.e., the time of greatest susceptibility). For perennial weeds, [CO₂] could stimulate greater belowground growth (e.g., rhizomes and roots), diluting the active ingredient of the herbicide. This latter possibility is seen for field-grown Canada thistle, where significant increases in belowground relative to shoot biomass with elevated [CO₂] were associated with increased herbicide tolerance.^[10]

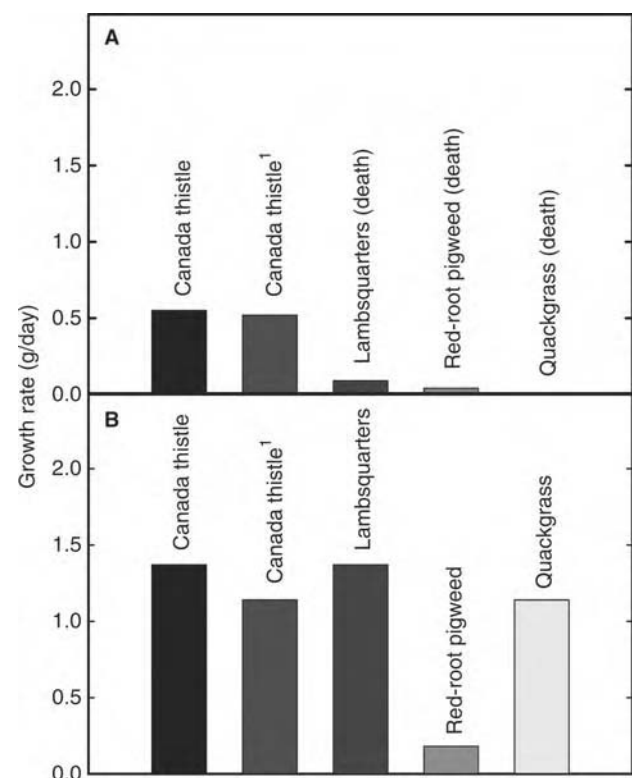


Fig. 2 Change in growth rate (g dry matter per day) for agronomic and invasive weeds when sprayed at recommended rates of herbicide at either (A) current [CO₂] or (B) future [CO₂] levels (600–800 ppm). All growth rates less than 0.1 resulted in plant death. Herbicide was glyphosate in all cases except Canada thistle,¹ which was sprayed with glufosinate. (From Ref.^[6].)

Biological Control

Rising [CO₂] could alter the efficacy of the biocontrol agent by altering the development morphology and reproduction of the plant host. In addition, [CO₂] would be likely to alter changes in the ratio of C:N and leaf protein levels with subsequent changes in the feeding habits and fecundity of insect herbivores. Overall, synchrony between plant development and the specific biological control agent is unlikely to be maintained as [CO₂] increases; however no specific data regarding [CO₂] and biological control agents are available, and a quantitative assessment is not yet possible.

Mechanical Control

A principal means of controlling weed populations is mechanical removal of the undesired plant. Tillage is regarded globally as a method of weed control in agronomic systems. However, elevated [CO₂] could lead to further below ground carbon storage with subsequent increases in the growth of roots or rhizomes, particularly in perennial weeds. Consequently, mechanical tillage may lead to additional plant propagation in a higher [CO₂] environment, with increased asexual reproduction from below ground structures and negative effects on weed control (e.g., Canada thistle). Nevertheless, as with biocontrol agents, no published studies are available regarding the interaction between rising [CO₂] and efficacy of mechanical control.

CONCLUSIONS

It is remarkable, given their importance in human systems, that so few data are available regarding the impact of [CO₂] on weed biology. As a result, extrapolation to in situ environments is difficult. Yet, given the current data, it is clear that the agricultural, environmental, and health costs of not understanding the impact of [CO₂] on weed biology may be substantial. It is hoped that the current article will serve to emphasize the critical nature of this topic and serve as an initial guide to those who wish to recognize the ramifications of rising [CO₂] beyond the polemic of global warming.

REFERENCES

1. Keeling, C.D.; Whorf, T.P. Atmospheric CO₂ records from sites in the SiO air sampling network. In *Trends: A Compendium of Data on Global Change*; Carbon Dioxide Information Analysis Center: Oak Ridge National Laboratory, U.S. Department of Energy, Oak Ridge, TN, 2001; 10–20.
2. Schimel, D.; Alves, D.; Enting, I.; Heimann, M.; Joos, F.; Raynaud, D.; Wigley, T.; Prather, M.; Derwent, R.; Ehrlert, D.; Fraser, P.; Sanhueza, E.; Zhou, X.; Jonas, P.; Charlson, R.; Rodhe, H.; Sadasivan, S.; Shine, K.P.; Fouquart, Y.; Ramasawamy, V.; Solomon, S.; Srinivasan, J.; Albritton, D.; Derwent, R.; Isaksen, I.; Lal, M.; Wuebbles, D. Radiative forcing of climate change. In *Climate Change 1995: The Science of Climate Change*; Houghton, J.T., Meira-Filho, L.G., Callander, B.A., Harris, N., Kattenberg, A., Maskell, K., Eds.; Cambridge University Press: Cambridge, U.K., 1996; 98–105.
3. Bowes, G. Photosynthetic responses to changing atmospheric carbon dioxide concentration. In *Photosynthesis and the Environment*; Baker, N.R., Ed.; Kluwer Press: Dordrecht, Netherlands, 1996; 387–407.
4. Robinson, A.; Robinson, Z. Science has spoken, global warming is a myth. *Wall Street Journal*, December 4, 1997.
5. Holm, L.G.; Plucknett, D.L.; Pancho, J.V.; Herberger, J.P. *The Worlds Worst Weeds. Distribution and Biology*; University of Hawaii Press: Honolulu, HI, 1977; 609 pp.
6. Ziska, L.H. Rising carbon dioxide and weed ecology. In *Weed Biology and Management*; Inderjit, Ed.; Kluwer Academic Publishers: The Netherlands, 2004; 159–176.
7. Ziska, L.H.; George, K. Rising carbon dioxide and invasive, noxious plants: potential threats and consequences. *World Resource Rev.* **2004**, *16*, 427–447.
8. Ziska, L.H. Climate change, plant biology and human health. *World Resource Rev.* **2003**, *15*, 271–287.
9. Ziska, L.H. Influence of rising atmospheric CO₂ since 1900 on early growth and photosynthetic response of a noxious invasive weed, Canada thistle (*Cirsium arvense*). *Funct. Plant Biol.* **2002**, *29*, 1387–1392.
10. Ziska, L.H.; Faulkner, S.S.; Lydon, J. Changes in biomass and root : shoot ratio of field-grown Canada thistle (*Cirsium arvense*), a noxious, invasive weed, with elevated CO₂: implications for control with glyphosate. *Weed Sci.* **2004**, *52*, 584–588.

West Nile Virus and Mosquito Control

David Pimentel

Department of Entomology, College of Agriculture & Life Sciences, Cornell University, Ithaca, New York, U.S.A.

INTRODUCTION

The West Nile virus, which causes serious encephalitis in Americans, was introduced from Africa into north-eastern United States in 1999. No one knows exactly how the virus was transported here, but with rapid air travel and large numbers of people and goods being moved throughout the world, the West Nile virus could have been carried to the United States by an infected bird, person, or even by a mosquito.

By the year 2003, the Centers for Disease Control (CDC) reported there were 8900 reported human infections of the West Nile disease with 218 deaths, with many of the infections and deaths occurring in Ohio. The rate of infections and deaths is running significantly ahead of last year, with most of the infections and deaths occurring in Colorado where the incidence has increased from only 14 infections in 2002 to 635 West Nile infections by August 2003.

BIRD RESERVOIRS

The prime reservoir of West Nile is the bird population. At least 125 species of birds have been reported infected with West Nile,^[1] with crows, blue jays, sparrows, hawks, eagles, and others identified as reservoirs. Birds appear to be especially susceptible to the virus and are more likely to die of an infection than are humans. In some localities crows and blue jays have all but disappeared. Estimates are that 20,000 birds were killed last year from West Nile in the United States. Because birds travel long distances in their seasonal migrations, infected birds spread the disease to humans, horses, and other animals. Mosquitoes obtain the virus mostly from infected birds and in turn infect humans by biting them.

MOSQUITO VECTORS

In the Northeast, the prime mosquito vector between birds and humans is *Culex pipens*, the house mosquito. In New York and New Jersey, when 32,000 mosquitoes were examined by the CDC,^[2] the great majority associated with West Nile were *Culex pipens*.^[3] In Colorado,

the prime mosquito vectors are *Culex tarsalis* and *Culex pipens*. Other mosquito species capable of transmitting the West Nile virus include other *Culex* species, *Anopheles* sp, *Coquilletidia* sp., *Ochlerotatus* spp., and *Psorophora* sp.

Male mosquitoes feed primarily on nectar and do not bite humans. The female mosquito requires a blood meal and when she bites an infected bird she then transmits the West Nile virus to humans by biting them.

The life cycle of *Culex* mosquito is about 14 days at temperatures of about 21°C (70°F). The female obtains her blood meal from birds, humans, and other animals. She mates either before or after her blood meal. Then she lays about 250 eggs in pools of water, including bird baths, flower pots, tin cans, old tires, as well as other pools of collected water. The egg stage lasts 1 to 2 days and the emerging larvae feed on algae, bacteria, and other organic matter in the water. The larval stage lasts 7 days followed by the pupal stage that lasts 2 to 3 days. Adult mosquitoes emerge from the pupae and the life cycle begins again. The adult mosquitoes normally live a week or two, but also hibernate in protected locations during the winter (Fig. 1).

Adult mosquitoes are not strong fliers and usually travel only a few hundred feet from the place of emergence. They may be carried by the wind several miles. In general, when the wind is blowing above 5 mph they will not fly. Female mosquitoes feed most often during the evening and morning.

MOSQUITO LARVAL CONTROL

The CDC advises that mosquito control should focus primarily on mosquito larval control and secondarily on the less efficient adulticiding.^[4] Effective larval control curtails the supply of adult mosquitoes.

In aquatic habitats, mosquito larvae have many predators, but few parasites. The predators include damselfly larvae, back swimmers, dragonfly larvae, water boatman, dytiscid beetles, frogs, fishes, and salamanders. However, none of these predators is effective because they usually inhabit permanent water bodies, whereas most mosquito larvae live in temporary pools of water.

Although mosquito larvae can be killed by bacteria, protozoans, nematodes, and fungi, none of these

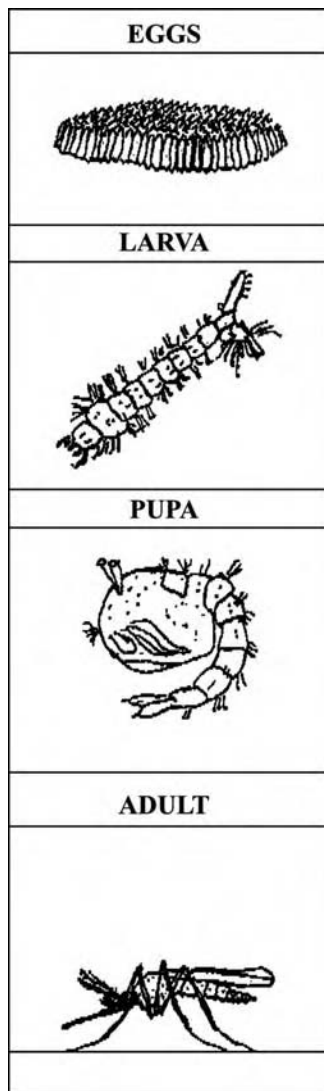


Fig. 1 *Culex* mosquito eggs, larva, pupa, and adult female.

provides control for large mosquito populations. One exception, a strain of *Bacillus thuringiensis israeliensis* or BT has proven effective. Various commercial formulations of this bacterium are available for application to ponds and pools where larvae are found.

In addition to eliminating all mosquito breeding sites, such as bird baths, flower pots, tires, ponds, and pools of water, such breeding habitats may also be treated, provided some water remains in them. BT is an effective larvicide that is safe for humans and pests, but it may kill some beneficial insects in water bodies.

In some small bodies of water, a thin layer of light oil can be spread over the surface. This will kill both mosquito larvae and pupae in the water. However, the oil also may have negative impacts on small fish and arthropods in the water.

Most insecticides are banned from water bodies because they are highly toxic to most aquatic organisms, such as fish, frogs, salamanders, and arthropods.

ADULT MOSQUITO CONTROL

Instead of focusing control efforts on larval mosquitoes as suggested by CDC, most homeowners and municipalities focus on adult mosquito control.

Adult Mosquito Control with Predators

Adult mosquitoes have relatively few predators because they are so small and not a large meal for a predator. Dragonflies, bats, and small birds such as purple martins feed on a few adult mosquitoes, but none of these animals can be counted on to control large populations of adult mosquitoes.

ULTRALOW VOLUME SPRAYING

Before municipalities spray for mosquitoes, the mosquito population should be measured for 5 days before spraying and 5 days after spraying using various mosquito traps. Such data will assist the government officials to determine whether the several thousand or millions of dollars spent in spraying was effective.

Homeowners should require warning 72 hr in advance of community spraying. During spraying, the windows and doors should be closed and the people should stay inside away from the insecticide spray.

When many West Nile infected birds are found and the mosquito population is relatively abundant, municipalities are often pressured into spraying pyrethroid insecticides for mosquito control. This spraying is carried out using trucks mounted with ultralow volume (ULV) sprayers. The insecticide spray produced from these units is like a smoke or fine mist and is carried downwind. Even assuming that the spraying is carried out in the evening when wind is minimal, the spray is carried downwind in an open area, for instance, on a golf course. Downwind, from 150 to 300 ft and at 3 ft height, the mosquito kill will range from 25% to 75%.^[5] However, ZERO mosquitoes will be killed upwind by the insecticide spray. Thus the average upwind and downwind kill is only 21% to 45%. Note, the insecticide spray does not penetrate buildings, and mosquitoes behind buildings are not killed. Further, dense vegetation hinders spray treatment and desired mosquito control. For example, downwind in a dense stand of trees, mosquito kill is reported to be only 34% to 58%.^[5]

For effective mosquito control, at least 90% of the adults must be killed. Only a few scientific studies of the effectiveness of spraying for mosquito control have been reported. These results are relatively discouraging. For example, in Greenwich, CT, only a 34% mosquito population reduction was reported after ground

spraying, and in Houston, TX, only a 30% reduction occurred after spraying.^[6] Then in Cicero Swamp, FL, populations of disease-carrying mosquito populations increased 15-fold after spraying,^[6] when the mosquito population was measured 11 days after spraying. However, it is doubtful that the insecticide spray caused the increase in the mosquito population, but clearly the insecticide provided insufficient adult mosquito control.

Aerial ULV Spraying

The aerial application of insecticides for adult mosquito control has some advantages over ground applications. Reports on the effectiveness of aerial ULV spraying range from 42% to 93%.^[7,8] However, using ULV aerial equipment results in only 10% to 25% of the insecticide reaching the target area, whereas up to 90% drifts away from the target into the environment at large.^[9,10] Aerial application covers a larger area faster than the ground application equipment, but it is more expensive than ground application, costing from \$250 to \$1000 per hour (truck spraying costs from \$150 to \$250 per hour). Also to be considered are the serious public health and environmental problems associated with the application of insecticides from aircraft.^[11]

Insecticide Effectiveness in Reaching Target Mosquitoes

With ULV spraying, the spray particles are minute and measure from 7 to 22 μm . The lethal dose of a pyrethroid insecticide is one particle 18 to 20 μm . Based on the fact that many billions of spray droplets are produced per kilogram of insecticide for both ground and aerial spraying, less than 0.0001% of the insecticide applied is reaching the target mosquitoes.^[12] Thus by both ground and aerial application 99.999% of the insecticide spreads into the environment, when it can cause public health and other environmental problems.

Because many adult mosquitoes remain after spraying and more adult mosquitoes will emerge, if the mosquito larvae are not controlled, then insecticide spraying is required every 7 days. Costs of spraying every 7 days are prohibitive.

PERSONAL PROTECTION

Homeowners should drain standing water in pools, gutters, and flower pots in the yard. Water in bird baths and wading pools should be changed every 3 days. If outdoors during dawn or dusk when mosquitoes are most abundant and the wind is not

blowing, then long pants and a long-sleeve shirt made of heavy material, such as denim, should be worn. Adult mosquitoes easily bite through a light T-shirt.

Various adult mosquito traps and zappers are sold to homeowners for control, but rarely do these units provide continuous satisfactory control of mosquitoes.^[13] While outside, homeowners may use an insecticide fogger or can of insecticide spray for temporary control of mosquitoes. However, if the wind is blowing sufficiently strong (5 mph or stronger), the mosquitoes will not be a problem because the mosquitoes will not fly in the wind.

Of the numerous chemical repellants, the most popular is the pesticide, DEET. DEET should be applied only to the outer layer of heavy clothes. The chemical should only be used, if there is a serious West Nile threat. DEET has been known to cause rashes, restlessness, lethargy, confusion, slurred speech, clumsiness, seizures, and in a few cases death.^[14] For some individuals, the DEET pesticide is reported to cause allergic reactions and may interfere with the immune and endocrine systems for some people.

Located on a patio or other small area, a large fan blowing air about 5 mph or higher will discourage the presence of mosquitoes.

CONCLUSION

West Nile virus is a health hazard to humans, birds, horses, and other animals. *Culex* mosquitoes are important vectors in the United States. The prime method of control is the elimination of the breeding habitats for larval mosquitoes, such as water accumulating in bird baths, flower pots, old tires, and other containers.

Widespread ULV spraying from ground equipment or aircraft for control of mosquitoes and West Nile virus is relatively ineffective, costly, and has been associated with environmental and public health risks.

During the evening and early morning, repellants can protect humans from mosquito bites. However, the pesticide DEET and related chemicals should not be applied directly to the skin of children or adults, because they pose serious public health risks.

REFERENCES

1. Environmental Defense. *West Nile Virus on the Rise, Threatening Humans and Wildlife*; Environmental Defense: New York, 2003. <http://www.environmental-defense.org/article.cfm?ContentID = 2871> (8/14/03).
2. CDC. *West Nile Virus: Statistics, Surveillance, and Control*; Centers for Disease Control: Atlanta, 2002. <http://www.cdc.gov/ncidod/dybid/westnile/surv&controlCaseCount02.htm> (8/17/03).

3. Nasci, R.S.; White, D.J.; Stirling, H.; Oliver, J.O.; Daniels, T.J.; Falco, R.C.; Campbell, S.; Crans, W.J.; Savage, H.M.; Lanciotti, R.S.; Moore, C.G.; Godsey, M.S.; Gotfried, K.L.; Mitchell, C.J. *Emerging Infectious Diseases*; Communicable Disease Center: Atlanta, 2001; Vol. 7. Past Issue. No. 4, Jul–Aug 2001. 10 pp.
4. West Nile Control. *West Nile Virus and Mosquito Control Practices*; 2002. <http://skipper.physics.sunyb.edu/mosquito/mosquito2/Mosquito2.htm> (8/16/03).
5. Mount, G.A. A critical review of ultralow-volume aerosols of insecticide applied with vehicle-mounted generators for adult mosquito control. *J. Am. Mosq. Control Assoc.* **1998**, *14* (3), 305–334.
6. Outcome. *Outcome Studies: Control Efforts for West Nile Virus and Mosquito Population*; 2003. http://www.cche-info.com/pdf/cche-wnv_outcome_studies.pdf (8/13/03).
7. Andis, M.D.; Sackett, S.R.; Carrol, M.K.; Bordes, E.S. Strategies for the emergency control of arboviral epidemics in New Orleans. *J. Am. Mosq. Control Assoc.* **1987**, *3* (2), 125–130.
8. Williams, R.E.; Knapp, F.W.; Clarke, J.L. Aerial insecticide applications for control of adult mosquitoes in the Ohio River Basin USA. *Mosq. News* **1979**, *39* (3), 622–626.
9. Bird, S.L.; Esterly, D.M.; Perry, S.G. Atmospheric pollutants and trace gases. *J. Environ. Qual.* **1996**, *25*, 1095–1104.
10. Pimentel, D.; McLaughlin, L.; Zepp, A.; Lakitan, B.; Kraus, T.; Kleinman, P.; Vancini, F.; Roach, W.J.; Graap, E.; Keeton, W.S.; Selig, G. Environmental and economic impacts of reducing agricultural pesticide use. In *Pesticide Question: Environment, Economics and Ethics*; Pimentel, D., Ed.; Chapman and Hall: New York, 1993; 223–278.
11. Pimentel, D. Environmental and economic costs of the application of pesticides in the U.S. In *Environment, Development and Sustainability*; *in press*.
12. Pimentel, D. Amounts of pesticides reaching target pests: environmental impacts and ethics. *J. Agric. Environ. Ethics* **1995**, *8* (1), 17–29.
13. Mosquito Buzz. In *Mosquito Magnet: Competitive Comparison Chart*; 2003. <http://www.mosquitobuzz.com/control/comparisonchart2.html> (8/16/03).
14. Marshall, L. Physicians urge caution with DEET. *Daily Camera* **2003**, 4A.

Wood Preservation

H. Michael Barnes

Forest Products Laboratory, Mississippi State University, Mississippi State, Mississippi, U.S.A.

INTRODUCTION

The history of wood preservation dates to 2000 B.C.E. when natural oils and other materials were used to preserve wood. Modern industrial timber preservation can be traced to John Bethell in England, who developed a process for pressure treating ship timbers with creosote in 1838.^[1] Today, wood preservation accomplishes two main tasks. First, it allows us to conserve timber. Experts estimate that the failure to control wood-destroying insects and fungi in the United States alone requires the additional cutting of 360,000 acres of forests yearly. Secondly, wood preservation allows us to increase the service life of wood. Treatment of wood affords protection from the principal agents of wood deterioration—fungi, wood-destroying insects (primarily termites), marine borers, fire, and weathering. A better understanding of the causal agents of wood deterioration will help the scientific community design more effective systems for protecting wood while minimizing environmental impact and improving service life. An excellent discussion of causal agents can be found in a recently published treatise.^[2]

In recent years, two principal factors have spurred changes in treatment technology and preservative systems worldwide: 1) environmental concerns, including air and water quality standards, and the effect of treated wood on man and nontarget organisms and 2) the energy crisis, especially in regard to oil and oil-based preservative systems. Of these two, environmental concerns predominate.

CLASSICAL WOOD PRESERVATIVES

Wood preservatives should be safe to handle and use, efficacious, cost-effective, and permanent, and should not corrode metal or degrade wood components. Worldwide, the major preservative systems are creosote, oilborne pentachlorophenol, and the waterborne arsenicals, primarily chromated copper arsenate (CCA) and ammoniacal copper zinc arsenate (ACZA). These systems have been designated Restricted Use Pesticides by the United States Environmental Protection Agency (EPA), but wood scraps and discarded components that have been treated with these preservatives are not listed as hazardous wastes. The major

concern with the disposal of treated wood is the lack of sanitary landfill space to accommodate a large volume of treated wood.

Creosote is a broad-spectrum biocide composed of a complex mixture of chemicals containing polyaromatic hydrocarbons, which can have immediate and chronic effects on exposed organisms. Fortunately, creosote is easily broken down in the environment and can be readily disposed of by high-temperature incineration. While creosote is an oil, it is often diluted with heavy oil or coal tar for use. It is primarily used to treat pilings or piles, poles, and crossties (sleepers).

Pentachlorophenol is a broad-spectrum biocide that is dissolved in organic solvents (often fuel oil). It is of concern because of its toxicity to aquatic organisms. It is banned in several countries and is strictly controlled in the United States. Pentachlorophenol is used primarily for treatment of crossarms and treatment of poles not exposed in tidal areas. Wood treated with pentachlorophenol can also be disposed of by high-temperature incineration.

The arsenic and/or chromium and arsenic in CCA and other waterborne systems can result in toxic reactions in aquatic organisms and pose an additional hazard because of their cumulative effect. Fortunately, such systems are well bound within the wood structure once it has been dried and fixed following treatment. A recent study showed that, while there were measurable biocide increases in the water column and sediment around treated wood in a wetland boardwalk, no taxa were excluded or significantly reduced near treated wood structures.^[3] Waterborne arsenicals are the primary preservatives used to treat lumber and timbers.

NEW GENERATION WOOD PRESERVATIVE SYSTEMS

Heavy metals like chromium and arsenic have undergone close environmental scrutiny spurring efforts to replace or reduce their use in waterborne systems. This has led to the development and introduction of several new copper-based preservative systems into the worldwide market. By agreement between the EPA and the wood preservation industry, wood treated with arsenical preservatives (CCA, ACZA) to retentions of 0.4 pcf or lower will be phased out and replaced with wood

treated with non-arsenical systems. These include copper-quaternary ammonium, chromated copper borate, copper azole, copper dimethyldithiocarbamate, bis-(*N*-cyclohexyldiazoniumdioxo)-copper, and copper citrate systems for aboveground and ground contact applications. The biocidal properties of borate compounds have long been used in Australasia and are coming into wider use in North America for wood exposed in protected, nonleaching environments, especially in areas threatened by the introduced Formosan subterranean termite. Borate formulations are being used in remedial treatment systems and zinc borate is being used for the protection of composite wood products. Borate-treated house framing components have been available in Hawaii for several years and now are available on the U.S. mainland.

New generation oilborne systems have also come to the fore in recent years. Many—such as substituted isothiazolones, chlorothalonil, thiazoles, carbamates, and triazoles—are under development or are in use as a component in multiple component preservative systems. Others, such as oxine copper and copper naphthenate, are re-emerging as commercial preservatives. Copper 8-Quinolinolate has United States Food and Drug Administration (FDA) approval for use in wood products in contact with foodstuffs (e.g., pallets) and is being used for preventing sapstain and mold fungi in freshly saw lumber, or for uses in aboveground exposures. Copper naphthenate in heavy oil carriers is finding use as a pole preservative and is not currently listed as a Restricted Use Pesticide by the EPA.

TREATING PROCESSES

Commercial pressure treating processes have remained largely unchanged since the early 1900s. Modifications to the standard practices include the modified full-cell process and the addition of a posttreatment fixation cycle with CCA preservatives. The modified full-cell cycle has been utilized to reduce the total solution injected into the wood while maintaining penetration and retention specifications. The net benefit is to reduce the potential release of excess preservative solution into the environment.^[4-6] Accelerated fixation cycles have improved compliance with environmental regulations and eliminated, or greatly reduced, post-treatment dripping. Rapid in situ fixation schemes with chromium and arsenic-containing preservatives generally use hot air heating, hot water fixation, steam fixation, or hot oil heating. The key factors affecting fixation are wood moisture content, temperature, concentration, and time. Since the fixation reactions are essentially ionic, moist wood is essential to proper rapid fixation.^[5]

Novel treating processes being developed may lead to improved treated products having reduced environmental impact. Among the emerging technologies are sonic treatment, gas/vapor-phase treatment, and super-critical fluid treatment. Of these, vapor-phase treatment using boron to treat composite materials is the closest to commercial use.

Detailed guidelines for best management practices have been issued for all major commercial wood preservatives in the United States.^[7] Use of these guidelines has been shown to reduce the impact of preservatives on the environment. Consumer Information Sheets have been issued for all wood preservatives to guide users in the proper use and handling of treated wood.^[7]

MANAGEMENT AND CONTROL OF PRESERVATIVE SYSTEMS

In the United States, preservative systems are managed and controlled by the Environmental Protection Agency, under the Federal Insecticide, Fungicide, and Rodenticide Act, and other governmental agencies through statutes designed to protect the environment. These long-standing statutes provide for air and water quality standards, discharge limits to the environment, certification, registration, remediation, and penalties for non-compliance.

In the European Union (EU), the Biocidal Product Directive was implemented in May 2000. Under its guidelines, those active biocidal ingredients, which are approved or in use in any of the member countries, must be listed and categorized as either “Identified” or “Notified” substances by March 2002. If a substance is considered “Identified,” its registration will be valid until 2005 or 2006. For the case of a “Notified” substance, data concerning human toxicity and its impact to environment must be provided by March 2002. In this case, and in the case of new biocide actives, full data, as required in the technical annex of the Biocidal Product Directive, must be provided for evaluation prior to approval for use. All wood preservative formulations will be registered at the member state level through existing national channels. It is intended that these registrations will be mutually recognized throughout the EU member states.

FUTURE CONSIDERATIONS

Many of the newer, more environmentally benign compounds suffer from a lack of broad-spectrum activity needed for ground contact application, or are highly leachable. This suggests that future systems will be based on combinations of narrow-spectrum biocides, similar to the co-biocide systems recently developed.

The use of multiple biocides seems especially cogent if the biocides act synergistically. Anchored biocides, which are covalently bonded to the wood, seem to offer another approach that would result in systems with lower depletion rates. To overcome energy-related problems, new organic preservatives requiring an oil-based carrier system need to be developed so that less oil carrier is needed.

REFERENCES

1. Graham, R.D. History of wood preservation. In *Wood Deterioration and Its Prevention by Preservative Treatments, Vol. I Degradation and Protection of Wood*, 1st Ed.; Nicholas, D.D., Ed.; Syracuse University Press: Syracuse, NY, 1973; 1–30.
2. Eaton, R.A.; Hale, M.D.C. *Wood: Decay, Pests, and Protection*; Chapman & Hall: London, 1993; 1–546.
3. Forest Products Laboratory. *Environmental Impact of Preservative-Treated Wood in a Wetland Boardwalk*; Research Paper FPL-P- U.S. Department of Agriculture Forest Service, Forest Products Laboratory, Madison, WI, 2000; 1–126.
4. Barnes, H.M.; Murphy, R.J. Wood preservation: the classics and the new age. *Forest Prod. J.* **1995**, *45* (9), 16–26.
5. Barnes, H.M. *Wood Protecting Chemicals for the 21st Century*, Int. Res. Grp. on Wood Pres. 1993, Doc. No. IRG/WP/93-30018, 1–28.
6. Wilkinson, J.G. *Industrial Timber Preservation*; Associated Business Press: London, 1979; 1–532.
7. Western Wood Products Institute. *Best Management Practices for the Use of Treated Wood in the Aquatic Environments, July 1996*; Vancouver, WA; www.awpi.org/wwpi/BMP/home.html (accessed November 3, 2000).

Worker Pesticide Exposure

Myna Panemangalore

Avinash M. Tope

Frederick N. Bebe

Department of Nutrition and Health, Land Grant Program, Kentucky State University, Frankfort, Kentucky, U.S.A.

INTRODUCTION

The commercialization of agriculture and the protection of crops to increase yields and quality have led to increased production and use of pesticides. Thus, occupationally exposed workers—those involved in the production and use of pesticides—have the highest risk of pesticide exposure, and manifest the greatest incidence of acute health effects or chronic illnesses such as neuropathy and cancer. The goal of this article is to provide a brief overview of various aspects of worker pesticide exposure and risk assessment.

PESTICIDE WORKERS

Pesticides, their residues, and metabolites are ubiquitous in the environment and are a major source of worker exposure. The two major categories of workers who are occupationally exposed to pesticides include agriculture/farm workers and factory workers.^[1] Farmers and farm workers (there are four million farm workers in the U.S.A.) constitute the largest group of exposed workers, and farm workers are mainly Hispanic young males (25–34 yr old). These migratory workers are less educated and poor, and most of them are located in the southern and western parts of the U.S.A. The demographics of these farm workers change between states every season/year.^[2]

PESTICIDE REGULATIONS FOR USE

The formulations used on crops contain the pesticide “active ingredient” (AI) and other compounds, “other ingredients.” Thus, the health effects of pesticides could be a consequence of either the AI or the “other ingredients.” The Environmental Protection Agency (EPA) evaluates all pesticides thoroughly as mandated by the Federal Insecticide Fungicide Rodenticide Act, and those that meet the requirements are then registered by EPA, which permits their distribution, sale, and use according to specific directions and requirements identified on the label. The various classes of

synthetic pesticides include organophosphates (comprises the largest group), organochlorines, carbamates, and synthetic pyrethroids. Other types of pesticides used include thiocyanates, and botanical pesticides such as nicotinoids, rotenoids, and pyrethrins.^[3] Fig. 1 shows the trend in the annual usage of pesticide AIs in agriculture, over a period of 20 yr. On an average about 70% of the pesticides produced in the U.S.A. are used for agriculture, and 17% for minor crops such as vegetables, fruits, and nuts, which comprise only 2% of the acreage planted.^[4] There are several reasons for the increase in pesticide use: 1) need to increase quality and yield of crops; 2) pest resistance to pesticides; 3) release of new pesticides to overcome resistance; 4) introduction of “no till” agriculture, which increases the use of herbicides; and 5) increase in the production of horticultural crops. The protection of pesticide workers and environmentally sound management of pesticide use is critical because large amounts of pesticides are used in agriculture.

HEALTH RISKS OF PESTICIDE EXPOSURE

The major problems with the use of pesticides are associated with their persistence in plants and the pollution of soil, water, and air, which constitute a major risk to non-target organisms. These include exposure/damage to farm animals and fish, which are important food sources, honey bees (essential for pollination of most crops), and human health, particularly in agricultural workers. The Food Quality Protection Act of 1996 requires EPA to set standards to protect the health of exposed and vulnerable populations, especially, children.^[3] Depending on use, exposure to pesticides varies in magnitude from insignificant to high levels that lead to either acute or chronic exposure. Acute exposure has immediate effects and causes overt toxicity and/or poisoning owing to multiple systemic effects, while chronic exposure (low-level exposure) results in health effects that become apparent years later. Farm pesticide exposure is generally cyclical, intermittent, and to multiple pesticides, whereas factory workers’ exposure is chronic and mostly to single pesticide or to the base

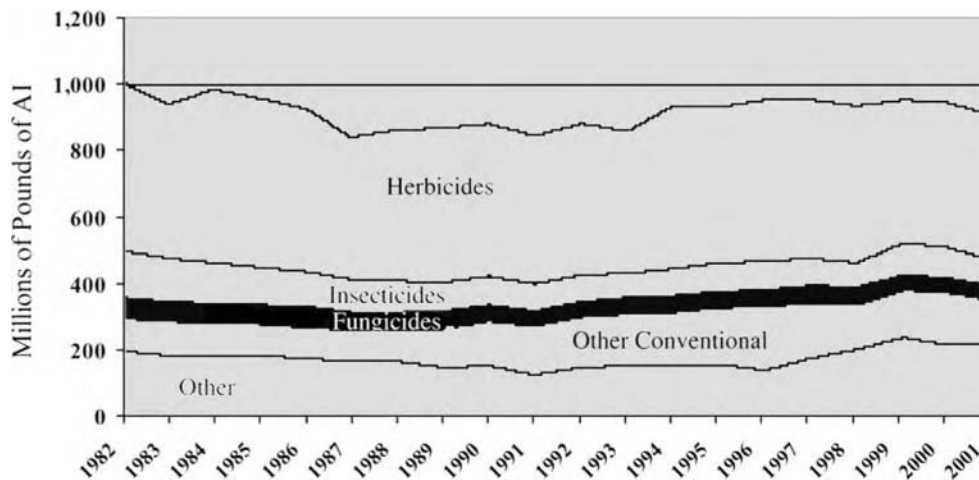


Fig. 1 The annual estimate of the amount of pesticide AIs used in the U.S.A. by pesticide type. (From Kiely, T.; Donaldson, D.; Grube, A. *Pesticide Industry Sales and Usage, 2000–2001 Market Estimates*; Biological and Economic Analysis Division, Office of Prevention, Pesticides, and Toxic Waste Substances, United States Environmental Protection Agency: Washington, DC, 2004; 28 pp.)

chemicals used to produce pesticides. Among farm workers, pesticide handlers (mixers, loaders, and sprayers) have the highest risk of exposure that can be acute or subchronic. However, it is important to point out that this kind of exposure has led to the development and use of personal protection equipment and guidelines for pesticide use. Even though directions for use and protection required are found on all pesticide containers, most small farm owners/workers neglect to use protective equipment/gear, and additionally, use spraying equipment without protective gear. Exposure to field workers and harvesters who work with sprayed agricultural crops results from pesticide residues remaining on the crops because of slow and/or inadequate decay of these compounds. Hand harvesting of crops (tobacco, fruits, and vegetables) increases the contact with pesticides residues. Tobacco workers in particular are exposed to pesticide residues and nicotine from wet tobacco leaf sap, which could lead to green tobacco sickness.^[5] It is necessary to educate farm workers to use measures such as wearing protective clothing, basic hygiene after spraying fields or field work, and consuming healthy diets with ample fruits and vegetables. Phytochemicals from fruits and vegetables provide protection against some of the adverse health effects of pesticides.

WORKER PROTECTION FROM PESTICIDE EXPOSURE

The US EPA and Health and Welfare Canada have produced a pesticide handler database for worker protection, which can be used to measure and standardize

exposure estimates.^[1] Toxicological studies are conducted to determine the Lethal Dose 50 (LD₅₀) in experimental animals (rodents) using oral or dermal pesticide exposure and include acute, subchronic, and chronic exposure. Dose response and LD₅₀ are used for risk assessment calculations to determine actual exposure dose in workers. Currently, many refinements are being considered for risk assessment of multiple pesticide exposure in workers who are intermittently exposed through dermal and inhalation routes.^[6]

TOXICOLOGICAL MANIFESTATIONS OF PESTICIDE EXPOSURE

In general, the harmful and/or toxicological manifestations of pesticides are related to exposure patterns, dose, frequency, and duration of exposure as well as the toxicodynamics of the pesticide, which vary among active compounds. Accidental poisoning can occur because of spills. The metabolic activation of pesticides by Phase I enzymes can produce reactive metabolites or electrophiles that could be more toxic and lead to the development of oxidative stress, which can in turn cause DNA damage and genotoxic changes such as chromosomal aberrations, micronuclei formation, and sister chromatid exchange.^[7,8] Chronic exposure to organophosphates and/or carbamates is known to lead to peripheral neurological damage because of the inhibition of acetylcholinesterase in blood.^[9] Comparatively, organochlorines tend to accumulate in adipose tissue and are metabolized more slowly than organophosphates. In acute exposure, the metabolites of organochlorines can stimulate the central nervous system and cause neurological problems and convulsions.^[10]

The symptoms of exposure in workers include headaches, burning eyes, muscular/joint pain, skin rashes, blurred vision, and shortness of breath, while chronic/prolonged exposure can depress brain function and cognition.

EPIDEMIOLOGICAL STUDIES AND RISK ASSESSMENT

Although epidemiological studies have limitations because of the complexities of multiple pesticide exposure that are exacerbated by smoking, and the lack of sensitive biomarkers in blood or urine, they provide important data that clearly suggest the adverse effects of pesticide exposure on human health and the development of chronic diseases. Many epidemiological studies in farm and greenhouse workers suggest a link between exposure to certain herbicides and organochlorines and development of non-Hodgkin's lymphomas. Some studies, but not all, indicate specific cancer risks and the development of neurological conditions at current occupational exposure levels. The ambiguity of epidemiological data may be a result of variations in the genetic susceptibility to pesticides and polymorphisms in the enzymes involved in the metabolism of these chemicals. More recently, epidemiological studies have been improved by considering factors that influence pesticide exposure, using integrated exposure measures such as exposure intensity and developing specific algorithms to predict pesticide exposure. There is a need to integrate data from human studies on health effects of pesticides with public health policy to lessen the impact of agricultural chemicals on health. Also a better understanding of the patterns of exposure and underlying variability within exposed groups, and better links between animal data and human health effects could improve evaluation of pesticide exposure risks.^[11]

CONCLUSIONS

The consideration of dermal and inhalation routes of exposure, smoking, age, integration of new data from metabolic, molecular, and genotoxic studies, and better designed epidemiological studies will permit a more accurate assessment for the health risks of occupationally exposed workers.

Furthermore, the identification and use of new biological pesticides, improvements in agricultural methods, pest ecology, and greater acceptance of genetically modified crops that improve pest resistance

could minimize the use of synthetic pesticides and reduce the risk of exposure and also their harmful effects in occupational farm workers.

ARTICLES OF FURTHER INTEREST

- Acute Human Pesticide Poisonings*, p. 3.
- Cancers from Pesticides*, p. 109.
- Hazard Labeling*, p. 1.
- Pesticide Sensitivities*, p. 606.
- Public Health and Costs of Pesticides*, p. 677.
- Reproductive and Developmental Effects from Occupational Pesticides Exposure*, p. 698.
- Worker Protection Standard*, p. 1.

REFERENCES

1. Kreiger, R.I. Pesticide exposure assessment. *Toxicol. Lett.* **1995**, *82/83* (1), 65–72.
2. United States Department of Agriculture. Website: http://www.usda.gov/wps/portal/lut/p/_s.7_0_A/7_0_1OB?q=farm+workers&num=10&mode=simple&navid=SEARCH&start=0 (accessed April 2005).
3. National Academies of Sciences. *The Future Role of Pesticides in US Agriculture*. National Academies Press: Washington, DC, 2000; 250–274.
4. Gianesse, L.P.; Marcelli, M.B. *Trend in Crop Pesticide Use; Comparing 1992 and 1997*; National Center for Food and Agriculture Policy, url: www.ncfap.org (accessed May 2002).
5. Ballard, T.; Ehlers, J.; Freund, E.; Auslander, M.; Brandt, V.; Halperin, W. Green tobacco sickness: occupational nicotine poisoning in tobacco workers. *Arch. Environ. Hlth.* **1995**, *50* (5), 384–389.
6. Hakkert, B.C. Refinement of risk assessment of dermally and intermittently exposed pesticide workers: a critique. *Ann. Occup. Hyg.* **2001**, *45* (1001), S23–S28.
7. Hodgson, E.; Levi, P.E. Pesticides: an important but underused model for environmental health sciences. *Environ. Hlth. Perspect.* **1996**, *104* (1), 97–106.
8. Panemangalore, M.; Dowla, H.A.; Byers, M.E. Occupational exposure to agricultural chemicals: effect on activities of some enzymes in the blood of farm workers. *Int. Arch. Environ. Hlth.* **1999**, *72*, 84–88.
9. Ray, D.E.; Richards, P.E. The potential for toxic effects of chronic low dose exposure to organochlorines. *Toxicol. Lett.* **2001**, *120*, 343–351.
10. Tordoir, W.F.; Sittert, N.J. Organochlorine. *Toxicology* **1994**, *91* (1), 51–57.
11. Alavanja, M.C.; Hoppin, J.A.; Kamel, F. Health effects of chronic pesticide exposure: cancer and neurotoxicity. *Annu. Rev. Pub. Hlth.* **2004**, *25*, 155–197.

Worker Protection Standard

James F. Ellerhoff

Iowa Department of Agriculture and Land Stewardship, Des Moines, Iowa, U.S.A.

Joyce S. Hornstein

Department of Entomology, Iowa State University, Ames, Iowa, U.S.A.

INTRODUCTION

Pesticides play a major role in increasing food production by reducing the number of crop-damaging pests, but exposure to pesticides can be harmful. The U.S. Environmental Protection Agency (EPA) reported that of the 1.2 billion pounds of pesticides used in the United States annually, 76% are used in agriculture. Farm workers and children are the primary population exposed to these pesticides.

BACKGROUND AND IMPLEMENTATION OF THE WPS

The EPA regulates pesticides under the Federal Insecticide, Fungicide, and Rodenticide Act (FIFRA). The Worker Protection Standard (WPS) for agricultural pesticides is a regulation issued by the EPA under the authority of FIFRA, which makes it unlawful for a person to use a pesticide in a manner inconsistent with its label. The WPS was first issued in 1974. In 1983, the EPA determined that hired farm workers were not being given adequate protection under their regulations. The EPA proposed revisions to the WPS, contained in Title 40, Part 170 of the Code of Federal Regulations, and solicited comments on the revisions. The final rule was adopted in August 1992, and published in the Federal Register and later in the “How to Comply” manual.^[1]

Implementation of the revised WPS emphasized educating agricultural employers on farms, forests, nurseries, and greenhouses about the revised regulation. The EPA and individual states developed and distributed WPS training materials and held presentations for employers to raise their awareness of their responsibilities to their pesticide handlers and workers. Because the WPS required label changes, the EPA also issued label amendment guidance to the pesticide manufacturers who registered their products.

REQUIREMENTS AND DEFINITIONS

The WPS is designed to protect applicators and workers in production agriculture. It does not apply to

pesticide uses in pastures, rangelands, homes, ornamental gardens, parks, and golf courses, or on livestock or other animals. Definitions of who is covered under WPS are helpful in understanding the requirements.

Employer. The employer is responsible for making sure that his or her employees follow the WPS. Employers provide the required personal protective equipment (PPE) and maintain it in good working condition, and ensure that the employees understand safety procedures. The employer must have a communication system in place so that field or greenhouse workers know when pesticide applications are made, when they can work with the crop, and what safety precautions to take. If a pesticide-related injury occurs, it is the employer’s responsibility to make sure that the worker receives medical care.

Worker. Workers perform tasks related to cultivation and harvesting of plants on farms, or in greenhouses, nurseries, or forests.

Handler. A handler is a person who mixes and makes pesticide applications.

Restricted-entry interval (REI). REI is the time period after a pesticide application when workers may not enter a pesticide-treated area without the protective clothing listed on the label.

Other definitions associated with the WPS are detailed in the “How to Comply” manual.^[1]

Duties for Employers

Pesticide safety training

Employees, including handlers and workers, must receive safety training every 5 years unless they are certified pesticide applicators. Training may be conducted by a certified applicator or by someone who has completed a train-the-trainer program. Details about the requirements can be obtained from state departments of agriculture.

Central location for pesticide information

Pesticide application information must be posted at a central location and the employer must tell workers

and handlers where this information is located and allow them access to it. The information includes:

- Product name, EPA registration number, active ingredient(s);
- Location and description of pesticide-treated areas; and
- Time and date of the application and length of the REI.

An EPA worker protection safety poster is included at the location along with the name, address, and telephone number of the nearest emergency medical facility.

Decontamination site

Employers must provide workers and handlers with water, soap, and single-use towels; handlers must be provided with a clean coverall. Emergency eyeflush water must be immediately available if the pesticide label lists protective eyewear.

Employer information exchange

An agricultural employer of commercial pesticide handlers must make sure that the customer (the operator of the farm, forest, nursery, or greenhouse) knows safety details about the pesticide applied. Similarly, the customer (operator) must communicate to the agricultural employer about pesticide-treated areas on the agricultural establishment where an application is to be made by a commercial handler.

Emergency assistance

An employer must promptly make transportation available to a medical facility if a handler or worker may have been poisoned or injured by pesticides. Information on the pesticide label, product use, and victims' exposure must be provided to medical personnel.

Anti-retaliation

Employers cannot retaliate against a worker or handler who attempts to comply with the WPS.

Additional Duties for Employers of Workers and Handlers

Workers must be kept out of areas being treated with pesticides. Typically workers are not to be in the pesticide-treated area during the REI. However, several exceptions are detailed in the "How to Comply"

manual^[1] and more recent amendments are on the EPA Web site.^[2] Workers must be notified about pesticide applications in their work area. Employers must either give oral or posted warnings and in some cases both. These warnings must be given to workers in a manner they understand, by using an interpreter if necessary.

The employer must make sure that handlers understand the information on the pesticide label. Handlers need to be instructed about the safe operation of equipment they are using. Commercial handlers must be made aware of areas where pesticides have been applied previously and whether there are any restrictions on entering those areas. Handler employers must make sure that pesticides do not contact people, other than trained and equipped handlers, during pesticide application. Monitoring must occur every 2 hr when a handler is using pesticides labeled with a skull-and-crossbones. Employers must provide handlers with PPE listed on the label and it must be clean, maintained in working condition, and fit correctly. This equipment must be stored in a clean place and the handler needs to have a place to store personal clothing. Employers must prevent heat-related illness while PPE is being worn.

Worker Protection Standard Updates

Because of the variety of agricultural practices, changes to the original WPS have been reviewed by the EPA and some have resulted in exceptions and exemptions to the original regulation. These changes include the following.

Crop advisors

While performing crop advisor tasks, certified or licensed crop advisors and persons under their direct supervision are exempt from most WPS provisions, except for pesticide safety training.

Limited contact exception

Under specified conditions, workers can enter pesticide-treated areas during the REI to perform tasks that involve limited contact with pesticide-treated surfaces.

Reduced REI for low-risk pesticides

The REI would be reduced from 12 to 4 hr for certain low-risk pesticides covered by the WPS.

Irrigation exception

Under specified conditions workers would be allowed to enter pesticide-treated areas during the REI to

perform irrigation tasks. This exception allows workers the flexibility during the REI to perform irrigation tasks that could not have been foreseen and which, if delayed, would cause significant economic loss. The exception includes provisions to limit pesticide exposure and risk to employees performing irrigation tasks during the REI.

Training requirements

The grace period (time before a worker must be trained) and the retraining interval for worker pesticide safety training were revised. This revision includes:

- A 5-day grace period for worker training.
- Agricultural employers must ensure that untrained workers receive basic pesticide safety information before they enter a treated area on the establishment. No more than 5 days after their initial employment has commenced, all untrained agricultural workers must receive the complete WPS pesticide safety training.
- The retraining interval for workers and handlers is 5 years.

Decontamination

This amendment reduces the number of days that decontamination supplies (soap, water, paper towels) are required to be available to workers after pesticide application. It applies to pesticides that are low risk and have REIs of 4 hr or less.

Warning signs

The size of the warning sign and the language requirements were changed to:

- Substitute the language commonly spoken and read by workers for the Spanish portion of the warning sign. The sign must be in the format required by the WPS and be visible and legible.
- Allow use of smaller signs provided that minimum letter size and posting distances are observed.
- Meet certain size and posting minimum requirements.

COOPERATION AMONG AGENCIES FOR EDUCATIONAL PROGRAMS, COMPLIANCE ASSISTANCE, AND EVALUATION

One of the difficulties for employers under the WPS is that the types of production agriculture across the

United States are diverse. States sought to adapt the WPS outreach programs to their particular agricultural activities. In Iowa, the state lead agency for pesticides is the Pesticide Bureau in the Iowa Department of Agriculture and Land Stewardship. The bureau is charged with the administration and enforcement of the Iowa Pesticide Act, Chapter 206 Iowa Code, and certain aspects of FIFRA through an annual cost-sharing grant with the EPA.

Iowa Survey

After several years of training, compliance assistance, and distribution of WPS-related publications,^[1,3] how well is the WPS understood by private pesticide applicators? We distributed a survey to more than 10,000 producers at training meetings held throughout Iowa in 1996. We asked general questions, including who handled and applied pesticides on the farm, their understanding of the WPS, their level of compliance, and the cost of compliance.

The findings were summarized^[4] as follows:

- Ninety-six percent indicated they themselves handled and applied pesticides, 29% hired custom applicators, and 23% indicated that other family members handled or applied pesticides.
- The private pesticide applicators rated their average level of understanding about the WPS as 6 (on a 10-point scale with 10 being a very high level). Of the questions asked, producers understood the requirements for family members better than those for non-family members. Posting pesticide information at a central location, displaying safety posters and emergency care information, and providing decontamination supplies were some of the least understood parts of the WPS. Survey respondents indicated their level of compliance with the WPS to be 7.
- Costs associated with WPS compliance were estimated to be \$235 per year. The top-ranked cost items are purchase of protective clothing, safety equipment, and time spent for education and training.

Michigan Survey

In a 1998 Michigan survey^[5] the intent was to determine how well the required WPS worker training was changing the pesticide safety knowledge of farm workers. Three years after training was mandated for all workers, only two-thirds of the survey participants said they had received training. Retention of much of

the training information ranged from minimal to approximately 50%. It is not clear whether the lack of retention was due to poor or no training, poor material, or language barrier.

FUTURE

To protect farmworkers, children, and pesticide applicators, employers must use up-to-date WPS safety training materials and programs. Individuals in contact with agricultural production areas need to receive this information. It is important to assess the background of these individuals so that effective communication takes place. The WPS program helps to ensure the safety of those individuals that work in farm fields, forests, greenhouses, and nurseries.

REFERENCES

1. U.S. Environmental Protection Agency. *The Worker Protection Standard for Agricultural Pesticides—How to Comply*; U.S. Government Printing Office: Washington D.C., 1993; 141 pp.
2. U.S. Environmental Protection Agency, www.epa.gov/pesticides/safety/workers/workers.htm (accessed August 2002).
3. Wintersteen, W.; Hornstein, J.S.; Miller, L. The worker protection standard: rules to improve pesticide safety. 1995; Iowa State University Extension, PM 1563b, 4 pp.
4. Hornstein, J.S.; Ellerhoff, J. Compliance with the worker protection standard. Proceedings of the 8th Annual Integrated Crop Management Conference; Iowa State University, 1996; 61–63.
5. Perry, S.; DiFonzo, C. The Worker Pesticide Knowledge Survey: Measuring the Success of Worker Protection Standard (WPS) Pesticide Safety Training, Final Report. Michigan State University, 1998; 1–20.

Index

- Abamectin (neonicotynl) insecticides, 129
 applications of, 475
- Acaricides, 265
- ACCase. *See* Acetyl coenzyme-A carboxylase.
- Acerophagus notativentris*, 211
- Acetaminophen, 349
- Acetolactate synthase (ALS), 170
- Acetolactate synthase (ALS)-inhibiting herbicides, 170
- Acetylcholine, 395
 and cholinesterase, reaction between, 69
 functions of, 399
 hydrolysis of, 395
 receptors
 nicotinic, 398–399
 postsynaptic, 399
- Acetylcholinesterase
 enzyme, 395–396
 inhibition of, 723
- Acetyl coenzyme-A carboxylase (ACCCase), 170, 344
- Acetyl coenzyme-A carboxylase (ACCCase)-inhibiting herbicides, 170
- Achatina fulica* (African snail), 33
- Achemon sphinx moth
 (*eumorpha achemon*), 207
- Ach hydrolysis, 69
- Acid detergent fiber (ADF), 324
- Acid detergent fiber (ADF), 324
- Acomys coharinus* (spiny mouse), 562
- Acrobasis nuxvorella*, 478
- Active ingredients (AIs), 157, 158, 722
- Acute or fast-acting rodenticides, 567–568
 fluoroacetamide (1081), 567
 sodium fluoroacetate (1080), 567
 strychnine, 567–568
 zinc phosphide, 568
- Acute toxicants (zinc phosphide), 528, 567, 695
- Acyrtosiphon pisum*, 11, 462
- ACZA. *See* Ammoniacal copper zinc arsenate.
- ADF. *See* Acid detergent fiber.
- Adjuvants
 activator, 1
 agricultural, 1
 and carriers, 1–2
 categories of, 1
 definition of, 1
 effects of, 2
 mode of action of, 1–2
 oil, 2
 oxyethylene, 2
 surfactants, 1
 toxicology of, 2
- [Adjuvants]
 traditional surfactant-type, 1
- Adsorption coefficient, definition of, 577
- Adsorptive endocytosis, 690
- Advance informed agreement (AIA), 364
- Aedes aegypti* (Asian tiger mosquito), 405
- Aedes albopictus*, 405
- Aerial ultra low volume (ULV),
 applications of, 4–6
- Aflatoxin, 87
 and aspergillus, 472
- African migratory locust (*locusta migratoria migratorioides*), 319
- African snail (*achatina fulica*), 33
- African stockpiles programme (ASP), 147
- AG. *See* Anastomosis groups.
- Agelaius phoeniceus*
 (red-winged blackbirds), 52
- Agricultural adjuvants, 1
- Agricultural crops, domestication of, 150–152
- Agricultural pesticides
 in developing countries, 626–629
 strategies for reducing risks
 with, 626–629
- Agricultural sustainability, 37
- Agrilus planipennis* (emerald ash borer), 654
- Agriotes obscurus* (dusky wireworm), 630
- Agrobacterium tumefaciens*, 199, 432
- Agrobiodiversity, 37
 functions of biodiversity in, 38
- Agrochemicals, mechanisms of
 resistance to, 344–346
 diagnosis of, 346
 impact of, 345–346
 metabolism, 344–345
 multidrug resistance, 345
 target site changes, 344
- Agroecosystems
 analysis, stages of, 293
 diversification of, 525
 European
 bioindicator development in, 38–39
 potential bioindicators for
 sustainability of farming
 practices in, 39
 functions of biodiversity in, 38
 hoverflies populations in, 248
 manipulation of, 360
 methods of biological weed
 control in, 276
 weed-crop interactions in, 712
- Agroenvironmental indicators,
 importance of, 38
- Agrostis stolonifera* (bentgrass), 305
- Agrotis ipsilon* (black cutworm), 109, 408, 461
- AIs. *See* Active ingredients.
- ALB. *See* Asian longhorned beetle.
- Aldicarb
 applications of, 597, 666
 intrinsic toxicity of, 665
- Alectoris chukar* (chukar), 347
- Aleochara bilineata*, 133
- Alepocurus mysuroides*
 (black grass), 344
- Alfalfa (*medicago sativa*)
 configuration of uncut, 323–324
 diseases
 control, considerations for, 9, 10
 development of, 7
 ecology and control of, 7–10
 effects of, 7–9
 types of, 8
 harvest manipulation, 322–324
 insects, ecology and management, 11–12
 integrated pest management in, 11
- Alfalfa mosaic virus (AMV), 7
- Alfalfa weevil (*hypera postica*), 114
 management of, 11
- Alkylphenol ethoxylates (APEs), 2
- Alkylphenols (APs), 2
- Allelochemicals, 360
- Allelopathy, 125
 role in weed suppression, 703
- Allolobophora caliginosa*, 49
- Alopex lagopus* (arctic fox), 347
- Alpha-chloralose (chronic rodenticides), 53, 568
- Alpha-endotoxin, 47, 49, 50
- Alpha-exotoxin (heat-labile), 553
- ALS. *See* Acetolactate synthase.
- Alternaria alternate*, 437
- Alternaria brassicae*, 56
- Alternaria brassicicola*, 56
- Amaranthus retroflexus* (pigweed), 125
- Amblyseius californicus*, 634
- Amblyseius reductus*, 631
- Ambrosia artemisiifolia* (ragweed), 58, 125
- Amendments, organic soil, 428–430
 effect of, 430
 efficacy of, 429
 impacts of, 428
 mechanisms of action, 429
 and plant resistance, 428
 use of, 429
- American serpentine leafminer (*liriomyza trifolii*), 462
- Ametadoria missella*, 211
- Amino acid biosynthesis inhibitors, 697

- Aminomethylphosphonic acid (AMPA), 220
 4-Aminopyridine, 54
 Ammoniacal copper zinc arsenate (ACZA), 719
 Amplified fragment length polymorphisms (AFLPs), 199
 AMV. *See* Alfalfa mosaic virus.
Anagrus atomus, 387
Anagrus epos, 387
Anagyrus clauseni, 211
Anagyrus subalbicornis, 211
Anaplophora chinensis (citrus longhorned beetle), 654
 Anastomosis groups (AG), 57
Anas wyvilliana (hawaiian duck), 349
 Anchored biocides, 721
Angropyron repens, 188
 Annihilation, male, 333
 Anilinopyrimidines, 77
 Animal and plant health inspection service (APHIS), 25
 Animal breeding, 14–16
 genetic resistance to disease, 14
 categories, 14
 methodology, for disease resistance, 14–16
 molecular biological methods, application, 15–16
 Animal genome maps, 16
 Animal pests
 monitoring of, 61
 in spring barley, 61
 in winter wheat, 61–62
 occurrence of, 60, 62
Anisopteromalus calandrae, 42
Anopheles gambiae, 350, 356
Anoplophora glabripennis (Asian longhorned beetle (ALB)), 21, 25–29, 653
 biology of, 21–23
 detection and control of, 23
 distribution and history of, 21
 ecology and control of, 21–23
 emergency program, 653
 impact of, 27
 abroad, 27
 invasion on North American urban forests, 25–29
 potential ecological disaster, 27
 Ant(s)
 in agricultural systems, role of, 114
 in crop insect control, 113–114
 fire
 attacks on animal and people, 183–184
 effects of, 183
 for pest control, 109
 roles in, crop insect control, 109–111
 social behaviour of, 114
 Ant–crop interactions, 110
Anthonomus grandis (cotton boll weevil), 333, 371
Anthonomus rubi (strawberry blossom weevil), 630
Anthonomus signatus (strawberry bud weevil), 630
 Antibiosis, 133, 198
Anticarsia gemmatalis nucleopolyhedrovirus (AgMNPV), 689, 690
 analysis of DNA of seasonal isolates of, 691
 electron micrograph of, 691
 Anticoagulants (chronic rodenticides), 568, 695
 Anticoagulant-type rodenticides, 528
 Antivenom, use of, 532
 Antixenosis, 133
 AOAC International. *See* Association of Official Analytical Chemists International.
Apanteles glomeratus, 133
Apanteles harrisinae, 211
Apanteles pedias, 683
Aphanomyces eutiches, 458
 Aphanomyces root rot, 458
 Aphid(s), 635–636
 black bean (*aphis fabae*), 428, 463
 black (*tinocallis caryaefoliae*), 478
 cabbage (*brevicoryne brassicae*), 385
 cowpea (*aphis craccivora*), 107, 463
 green peach (*myzus persicae*), 345, 435, 463, 635
 melon (*aphis gossypii*), 114, 248, 435, 595, 635
 mustard (*lipaphis erysimi*), 598
 pecan (*monellia caryella*), 478
 potential control of, 247–248
 soybean (*aphis glycinis*), 654
 strawberry (*chaetosiphon fragaefolii*), 630–631, 635
 sustainable farming and potential control of, indicators of, 247–250
 syrphidae as potential control of, 247–248
 wheat straw mulch in repelling, 20
Aphidius smithi, 322
Aphis craccivora (cowpea aphid), 107, 463
Aphis fabae (black bean aphid), 428, 463
Aphis glycinis (soybean aphid), 654
Aphis gossypii (melon aphid), 114, 248, 435, 595, 635
Aphytis maculicornis, 427
Apis mellifera, 5, 536
 Apple sign test, 693
 Aquatic weeds
 chemical control, 640–641
 in flowing water, 641
 in New Zealand lakes, 641–642
 in recreational lake, 641
 mechanical control
 cutting and harvesting, 637
 draglining/dredging, 638–639
 rototilling, 638–639
Arachis hypogaea (peanut) diseases
 cylindrocladium black rot, 470
 ecology and control of, 469–473
 groundnut rosette, 470
 leaf spots, early and late, 469
 rust, 469–470
 stem rot, 470
 Arboviral disease, 141. *See also* Dengue.
 Arctic fox (*alopex lagopus*), 347
Arilus cristatus, 536
 Arthropod(s)
 associated to papaya, 441–443
 [Arthropod(s)]
 pest, 474
 of cherry, 79
 control, use of IPM, 686
 of grape, 207
 management of, 686
 of strawberries, 630–632
 predaceous, impact on, 114
 strawberry
 ecology and control of, 630–631
 insects and mites in California, ecology and control, 634–636
 Arthropod mass rearing and quality control working group (AMRQC), 382
 Arthropods, poisonous, 531–537
 apidae. bees, 536
 arachnida, 531–534
 scorpions, 531
 spiders, 532–533
 ticks, 533–534
 chilopoda, centipedes, 534
 diplopoda, millipedes, 534
 formicidae. ants, 535–536
 hemiptera. true bugs, 536
 hymenoptera, 535
 insecta, 535–537
 lepidoptera. caterpillars and moths, 534–535
 Artificial diets, 370–372
 for predators and parasitoids, 370–371
 quality control of natural enemies produced on, 371
 successes and failures with, 371
Aschrysoophagus modestus, 683
 Ascochyta blight complex (mycosphaerella blight), 457
 life cycle of, 458
 Ash whitefly (*siphoninus phillyreae*), 299
 Asian ladybird beetle (*harmonia axyridis*), 245
 Asian longhorned beetle (ALB) (*anoplophora glabripennis*), 21, 25–29, 653
 biology of, 21–23
 detection and control of, 23
 distribution and history of, 21
 ecology and control of, 21–23
 emergency program, 653
 impact of, 27
 abroad, 27
 invasion on North American urban forests, 25–29
 potential ecological disaster at home, 27
 Asian tiger mosquito (*aedes aegypti*), 405
 Aspergillus and aflatoxin, 472
Aspergillus flavus, 87, 472
Aspidiotus nerii (oleander scale), 425
 Association of Natural Bio-control Producers (ANBP), 382
 Association of Official Analytical Chemists International (AOAC International), 550
Athelia rolfsii, 470
Atherigona soccata (shoot fly), 618
Atractotomus mali, 173
 ATSO®, 420
 Attract and kill trap, 426

- Australian plague locust
(*chortoicetes terminifera*), 319
- Autographa californica*, 690
- Auxin transport inhibitor, 136
- Avermectin, 398
- Avicides, 52
- Avigrease[®], 52
- Avis Scare[®], 52
- Avitrol[®], 52
- Azadarachta indica* (meliaceae), 394, 428, 688
- Azocosterol (Ornitrol[®]), 54
- Azoxystrobin, 647
- Azteca chartifex*, 110
- Bacillus sphaericus*, 32, 354, 358
- Bacillus subtilis*, 215
- Bacillus thuringiensis* (Bt), 31–32, 197, 272, 394, 479, 523, 659, 716
culture and control of, 31–32
formulations of, 310
toxins, 34, 45
effects, 34
transgenic plants, resistance, 553–554
- Bacillus thuringiensis israelensis* (Bti), 31, 354, 358
- Bacillus thuringiensis tenebrionis*, 31
- Bacteria
canker, 76–77
diseases, 623
management of, 77
symptoms, 77
diseases, 437
entomopathogenic, 659–660
pest controls, 30–32
biological agents, potential, 32
- Bactrocera cucurbitae* (melon flies), 440
- Bactrocera dorsalis* (fruit flies), 440
- Bactrocera oleae* (olive fly)
life history of, 425
management of, 425–426
monitoring of, 425
- Bactrocera papayae*, 440
- Baculovirus anticarsia*, production and
formulation of bioinsecticide, 690
- Baculoviruses, 689
activity in host insects, 692
commercial production of, in insect
cells, 692
genetic modification of, 692
host specificity of, 691–692
pathogenesis, 689, 690
phylogeny of, 691
populations of, 691
use of, 689
- Baits, poison
for arthropods
advantages and limitations, 527
composition, 527
efficacy, 527, 528
future needs, 528–529
for rodents
advantages and limitations, 528
composition and distribution, 527–528
efficacy, 529
- [Baits, poison]
selectivity and attraction of, 528
- Baits, chronic, 528
- Banana weevils (*cosmopolites sordidus*), 110
- Bancroftian filariasis, 179
- Bandicoot rat (*bandicota bengalensis*), 562
- Bandicota bengalensis* (bandicoot rat), 562
- Barn owl (*tylo alba*), 33
- Barriers for vertebrate pest
bats, 513
birds, 511
opossums, 513–514
rabbits, 514
raccoons, 513–514
rodents, 511–513
skunks, 513–514
white-tailed deer, 514–515
- Basidiomycetes, 662
- Bayrepel[®], 358
- Beauveria bassiana*, 268, 454
- Beetle(s)
Asian ladybird, 245
- Asian longhorned
biology of, 21–23
detection and control of, 23
distribution and history of, 21
ecology and control of, 21–23
emergency program, 653
impact of, 27
invasion on North American urban
forests, 25–29
potential ecological disaster
at home, 27
- brown spruce longhorn, 290
- carbid, 361
- cereal flea, 61
- citrus longhorned, 654
- coconut rhinoceros, 92, 93
- colorado potato
biological cycle, seasonal, 100
control of, 100
dispersal of, 256
impact of heat on, 99
insecticide resistance in, 272
mortality rates of, 100
resistance in, 272
thermal control of, 99–101
visual index damage, 99
- Japanese, 30
- lema
economic importance of, 446
as host of parasites, 446, 447
occurrence of, 447
parasites on, 446–448
- olive bark, 425
- shoot, 290, 654
- twig cutter, 425
- Beet pseudo yellows virus (BPYV), 635
- Beet webworm (*loxistege sticticalis*), 461
- Bemisia argentifolii* (silverleaf whitefly), 652
host for, 606
incidence of, 608
infestations of, 607
management of, 606–609
response, to reflective plastic and wheat
straw mulch, 606, 608
cucumber, 606–607
- [*Bemisia argentifolii* (silverleaf whitefly)]
pumpkin, 606
spectral reflectance, 608
squash, 608
symptoms of, 606
- Bemipidion lampros* (carbid beetle), 361
- Benzenehexachloride (BHC), 70
- Benzimidazoles, 344
and thiophanates, 73
- Bermuda grass (*cynodon dactylon*),
188, 303
- Beta-exotoxin (heat-stable), 553
- BHC. *See* Benzenehexachloride.
- Bioaccumulation, 385
effects of, 539
process of, 538
- Bioassay, 346
techniques, 5–6
- Biocide, 719
anchored, 721
creosote, 719
pentachlorophenol, 719
- Biocontrol
agents
effects of, 33, 35
role of, 35
augmentation, 173
conservation, 173
effects of, 33
forms of, 33
health risks, human, 34
limits of, 33–36
methods, 34
genetic modification, 34
types of, 34
mitigating risk of, 35
risk from biochemicals, 34
risks associated with, 33
risk to non-target species, 33–34
- Biodegradation, pesticides, 161–162
- Biodiagnosis, 492
- Biodiversity
in agroecosystems, functions of, 38
conservation of, 248
hoverflies as indicators of, 248–249
- Bioherbicides, efficacy of, 193
- Bioindicators, 37–40
development in, 38–40
coffee in latin America, 38–40
European agroecosystems, 38
for sustainability of farming practices
examples, 39
future studies on, 40
- Bioinsecticides, 689
production and formulation of, 690
- Bioinvasions, 34, 540
risk of, 34
- Biological control agents (BCAs), 675
- Biological weed control, principal
methods of, 275–276
- Biomagnification, 538, 539
- Biomarkers in blood or urine, lack of
sensitive, 724
- Biopesticides, use of, 159
- Bioreactors, 692
- Biosynthesis inhibitors
amino acid, 697

- [Biosynthesis inhibitors]
 ergosterol, 73, 215
 lipid, 697
- Biotechnology
 assessment of transgenic virus-resistant
 potatoes in Mexico, 47–50
 disease resistance in crops, 45–47
 genetic engineering in pest control,
 benefits, 45
 for insect control, 49–50
- Bioterrorism, 402
- Bipolaris sacchari*, 643
- Bird control
 aspects of lethal, 54
 chemicals, 52–55
 immobilizing agents, 53
 lethal stressing agents, 53
 repellents, 54
 reproductive inhibitors, 54
 toxicants, 52
 nonlethal methods of, 54
- Bird reservoirs, 715
- Bird Shield[®], 54
- Bixadus sierricola*, 96–97
- Black aphid (*tinocallis caryae-foliae*), 478
- Black bean aphid (*aphis fabae*), 428, 463
- Black cutworm (*agrotis ipsilon*), 109,
 408, 461
- Black flat mite
 (*brevipalpus phoenicis*), 444
- Black grass (*alepocurus mysuroides*), 344
- Black rot pathogen (*ceratocystis
 fimbriata*), 655
- Black scale (*saissetia oleae*), 425, 426–427
- Black sigatoka, in banana, 573, 574
- Black vine weevil (*otiorhynchus
 sulcatus*), 631
- Blissus leucopterus leucopterus*
 (chinch bugs), 618
- Blue butterfly (*maculina arion*), 35
- Blumeriella jaapii* (*cocomyces hiemalis*), 75
- Boisea trivittatus* (boxelder bug), 245
- Boll weevil attract and control tube
 (BWACT), 333
- Bombus occidentalis* (bumblebees), 453
- Borate compounds, biocidal
 properties of, 720
- Botrytis cinerea* (gray mold), 214–215, 459
- Bovine leukosis, 14
- Boxelder bug (*boisea trivittatus*), 245
- BPH. *See* Brown plant hoppers.
- BPYV. *See* Beet pseudo yellows virus.
- Brachymeria intermedia* (chalcid wasp), 370
- Bracon cushmani*, 211
- Braconid wasp (*cardiochiles nigriceps*), 114
- Branchinella kugenumaensis*, 40
- Branta canadensis* (Canada geese), 54
- Brassica napus* (canola), 169
- Brassica oleracea* (cabbage) diseases, 56–59
 black rot, 56
 black spot, 56–57
 clubroot, 56
 control principles, 57–58
 dark leaf spot, 56–58
 downy mildew (*peronospora viciae*), 56
 ecology and control of, 56–59
 management practices for, 57
- [*Brassica oleracea* (cabbage) diseases]
 pathogens ecology, 56
 foliar, 58–59
 seedborne, 58
 soilborne, 58
 sclerotinia stem rot, 57
 therapy, 58
 watery soft rot, 57
 white mold, 57
 wirestem, 57
- Breeding
 genetics of resistance and plant, 197–199
 mutation of, 198
 plant, 519–523
 strategies for, 522
 for resistance, 197
 strategies, 198
- Breeding, animal, 14–16
 genetic resistance to disease, 14
 categories of, 14
 methodology, for disease resistance, 14–16
 molecular biological methods,
 application, 15–16
- Bremia lactucae*, 317
- Brevicoryne brassicae*
 (cabbage aphid), 385
- Brevipalpus phoenicis*
 (black flat mite), 444
- Broad mite (*polyphagotarsonemus
 latus*), 444
- Bromethalin (chronic rodenticides), 568
- Bromus tectorum* (downy brome), 126
- Brown locust (*locustina pardalina*),
 319–320
- Brown plant hoppers (BPH)
 (*nilaparvata lugens*), 293, 539,
 552, 597
- Brown spruce longhorn beetle
 (*tetropium fuscum*), 290
- Brown treesnakes (*boiga irregularis*), 347
- Bruchus pisorum* (pea weevil), 463
- Brugia malayi*, 179
- Brugian filariasis, 179, 180
- Brugia timori*, 179
- Brush-tail possums (*trichosurus
 vulpecula*), 349
- Budded viruses (BVs), 689, 690
 dissolution of, 689
- Bufo marinus* (giant toad), 347
- Bugs
 boxelder, 245
 chinch, 618
 cocaine, 139
 lygus, 322–324
 mealy, 200, 504, 696
 pirate, 621
 western tarnished plant, 106, 631, 635
- Bulbuls (*pycnonotus jocosus*), 347
- Bumblebees (*bombus occidentalis*), 453
- Bursaphelenchus cocophilus*, 91
- Butterfly, blue (*maculina arion*), 35
- Butterfly, monarch (*danaus plexippus*), 255
- Butterfly, white (*peris brassicae*), 132
- Buzz-Off[®], 54
- BVs. *See* Budded viruses.
- BWACT. *See* Boll weevil attract and
 control tube.
- Cabbage aphid (*brevicoryne brassicae*), 385
- Cabbage (*brassica oleracea*) diseases, 56–59
 black rot, 56
 black spot, 56–57
 clubroot, 56
 control principles, 57–58
 dark leaf spot, 56–58
 downy mildew (*peronospora viciae*), 56
 ecology and control of, 56–59
 management practices for, 57
 pathogens ecology, 56
 foliar, 58–59
 seedborne, 58
 soilborne, 58
 sclerotinia stem rot, 57
 therapy, 58
 watery soft rot, 57
 white mold, 57
 wirestem, 57
- Cabbage looper (*trichoplusiani*), 598
- Cactoblastis cactorum* (South American
 moth), 34
- Calacarus brionese* (papaya leaf edge
 roller mite), 444
- Calciferol (vitamin D)
 (chronic rodenticides), 568
- Calepitrimerus vitis*, 200
- Callosobruchus maculatae*
 (cowpea weevil), 463
- Callosobruchus maculatus*
 (cowpea bruchid), 106
- Calonectria ilicicola*, 470
- Caloptilia theivora* (gracillariid leaf
 roller), 672
- Camellia assamica*, 672
- Camellia sinensis*, 672
- Campylomma verbasci*, 173
- Canada geese (*branta canadensis*), 54
- Canker
 bacterial, 76–77
 management of, 77
 symptoms of, 77
 disease, citrus bacterial, 517, 518
 eradication program, 517
 perennial, 467–468
- Canola (*brassica napus*), 169
- Capnodis tenebrionis*, 81
- Capsaicin, 694
- Capsicum chinense*, diversity in, 151
- Captive breeding, paradox of optimization
 strategy, 175–177
- Captive population, establishing and
 maintaining of, 177
- Captive rearing, insects
 for field release, 175–178
 paradox of, 175
- Carambola fruit borer
 (*eucosma notanthes*), 502
- Carbamate(s), 252, 398
 dithio, 73
 herbicides, 72
 immunotoxic effects in man, 252
 insecticide and ChE enzyme, reaction
 between, 70
- Carbid beetle (*bempiidion lampros*), 361
- Cardiochiles nigriceps* (braconid wasp), 114
- Carfentrazone-ethyl, 135

- Carica papaya* (papaya)
 apical necrosis, 436
 arthropods associated to, 441–444
 droopy necrosis, 436
 fruit flies, 440, 441
 insects
 ecology and control of, 440–445
 sampling and monitoring of, 440
 yellow-type diseases in, 436
- Carica papaya* (papaya) diseases
 bacterial, 437
 ecology and control of, 435–438
 fungal
 alternaria fruit spot, 437
 anthracnose, 437
 cercospora black spot, 437
 control of, 438
 dry rot, 437
 fruit rot and root rot,
 phytophthora, 437
 fusarium fruit rot, 437
 internal blight, 437
 powdery mildew, 437
 nematode-borne
 reniform, 436
 root knot, 436
 phytoplasma, 436
 viral
 meleira or sticky, 435–436
 mosaic, 435
 ring spot, 435
- Carposina sasakii* (peach fruit moth), 474
Carya illinoensis (pecan), 478
 Cat fleas (*ctenopthalides felis*), 405
Catolaccus grandis, 371
 CBR. *See* *Cylindrocladium* black rot.
 CCA. *See* Chromated copper arsenate.
 CDC. *See* Centers for disease control.
Cemistoma coffeella, 95
Cemistoma coffeellum, 95
 Center-pivot irrigation systems, 64
 Centers for disease control (CDC), 715
Cephus cinctus (wheat stem sawfly), 408
Ceratitis capitata (medflies), 440, 652
Ceratocystis fimbriata
 (black rot pathogen), 655
Ceratocystis paradoxa
 (pineapple sett rot), 644
Ceratophyllum demersum (hornwort or
 coontail), 639
Cercospora arachidicola, 469
Cercospora papayae, 437
Cercosporidium personatum, 469
 Cereal flea beetle (*phyllostreta vittula*), 61
 Cereals, 60
 growing systems of, 60–62
 pest occurrence of, 60–62
Ceriagrion melanurum, 40
Ceroplastes sinensis (chinese wax scale), 661
Cervus elaphus (red deer), 347
 CFF. *See* Cherry fruit flies.
 CfMNPV. *See* *Choristoneura fumiferana*.
 CFR. *See* Code of federal regulations.
Chaetosiphon fragaefolii
 (strawberry aphid), 630–631, 635
 Chalcid wasp
 (*brachymeria intermedia*), 370
Chamaesyce humistrata
 (prostrate spurge), 303
Chaoborus astictopus, 538
Chara australis, 640
 ChE enzyme, 69
 carbamate insecticide and, reaction
 between, 70
 Chemigation, 63–66
 advantages and disadvantages of, 65
 concept of, 66
 as IPM tool, 63–64
 use of, 65
Chenopodium album, 125
 Cherry
 insects, ecology and control of, 79–86
 pests, 79, 85
 arthropod, 79
 control of, 85
 management of, 85
 production, 80–82
 sweet, 75, 79
 tart, 75, 79
 Cherry diseases
 bacterial canker, 76–77
 management of, 77
 symptoms of, 77
 ecology and control of, 75–78
 fungal, 75–76
 brown rot, 75
 leaf spot, symptoms of, 75–76
 powdery mildew, 76
 management of, 77
 viral, 77
 Cherry fruit flies (CFF), 80
Cheyletus eruditus, 43
 Chilopoda. centipedes, 534
 Chinaberry (*melia azedarach*), 416
 Chinch bugs (*blissus leucopterus
 leucopterus*), 618
 Chinese wax scale (*ceroplastes sinensis*), 661
 4-Chloro-2-methylphenoxyacetic acid
 (MCPA), 253
 Chloronicotiny, 300
 Chlorophenoxy acids, 72
Chlorops pumilionis (straw fly), 61
 Cholecalciferol (vitamin D₃)
 (chronic rodenticides), 568
 Cholinesterase
 and acetylcholine, reaction between, 69
 inhibition of, 69
 in vitro, inhibitors of, 69
 model of, 69
Choristoneura fumiferana (CfMNPV), 689
Chortoicetes terminifera (Australian plague
 locust), 319
 Chromated copper arsenate
 (CCA), 719
Chromatomyia horticola
 (pea leafminer), 462
 Chromototropic trap, 247
 Chronic baits, 528
 Chronic diseases, development of, 724
 Chronic (slow-acting) rodenticides
 alpha-chloralose, 568
 anticoagulants, 568
 bromethalin, 568
 calciferol (vitamin D), 568
 Chronic toxicants, 528
Chrysodeixis eriosoma, 600
Chrysopa californica, 453
Chrysops dimidiatus, 181
 Chukar (*alectoris chukar*), 347
 CIPAC. *See* Collaborative International
 Pesticide Analytical Council Ltd.
 Citrus bacterial canker disease, 517, 518
 eradication program, 517
 Citrus longhorned beetle (*anaplophora
 chinensis*), 654
 Citrus mealybug (*planococcus citri*), 504
Cladosporium carpophilum, 468
Cladosporium cladosporioides, 459
Clavigralla tomentosicollis, 106
 CLI. *See* Consumer labeling initiative.
Clostridium botulinum, 297
 Cluster caterpillar (*spodoptera litura*), 333,
 504, 631
 Cluster fly (*pollenia rudis*), 245
 Coat protein (CP) gene, 435
 Cocaine bugs, 139
Coccinella septempunctata, 456
Coccomyces hiemalis (*blumeriella jaapii*), 75
Coccophagoides utilis, 427
 Cochliomyia hominivorax, 484
 Coconut (*cocos nucifera*)
 insects, ecology and control, 90–91
 pests, 90–92
 Coconut rhinoceros beetle
 (*oryctes rhinoceros*), 92, 93
Cocos nucifera. *See* Coconut.
 Code of federal regulations (CFR), 495
 Codling moth (*cydia pomonella*) (CpGV),
 474, 689
 Coffee berry borer
 (*hypothenus hampei*), 96
 Coffee insects, ecology and control of, 95
 Coffee leaf miner (*leucoptera coffeella*),
 95–96
 Coffee stem borers, 96–97
 management techniques, 97
Coleomegilla maculata, 621
Coleomegilla maculate, 408
 Colibacillosis, 14
 Collaborative International Pesticide
 Analytical Council Ltd.
 (CIPAC), 550
Colletotrichum gloeosporioides, 437
Colletotrichum pisi, 459
 Colloid-osmotic lysis, 553
 Colorado potato beetle (CPB) (*leptinotarsa
 decemlineata*), 665
 biological cycle, seasonal, 100
 control of, 100
 dispersal of, 256
 impact of heat on, 99
 insecticide resistance in, 272
 mortality rates of, 100
 thermal control of, 99–101
 efficacy of, 99–100
 visual index damage, 99
Columba livia (rock pigeon), 52
 Conditioned taste aversion (CTA), 567
Coniothyrium minitans, 649
Conopomorpha sinensis
 (litchi fruit borer), 504

- Consultative Group on International Agricultural Research (CGIAR), 487
- Consumer labeling initiative (CLI), 495
- Control-A-Bird[®], 52
- Coontail or hornwort (*ceratophyllum demersum*), 639
- Cooperative Agriculture Pest Survey (CAPS) program, 653
- Copper fungicides, 670
- Copper 8-quinolinolate, 720
- Copidosoma floridanum*, 600
- Corn earworm (*helicoverpa zea*) (HzSNPV), 408, 689
resurgence of, 597
- Corn leaf hopper (*dalbulus maidis*), 114
- Cosmopolitan, commensal species, 562
- Cosmopolites sordidus* (banana weevils), 110
- Costelytra zealandica* (New Zealand grass grub), 31
- Cotesia glomerata*, 525
- Cotton boll weevil (*anthonomus grandis*), 333, 371
- Cowpea aphid (*aphis craccivora*), 107, 463
- Cowpea bruchid (*callosobruchus maculatus*), 106
- Cowpea seed moth (*cydia ptychora*), 106
- Cowpea (*vigna unguiculata*) insects
ecology and control of, 106–108
pests, 106–107
 coleoptera, 106
 heteroptera, 106–107
 lepidoptera, 106
 management of, 107
 thysanoptera, 107
- Cowpea weevil (*callosobruchus maculatae*), 463
- CPB. *See* Colorado potato beetle.
- CpGV. *See* *Cydia pomonella*.
- Crataegus monogyna*, 388
- Creosote, 719
- Cropping systems
negative effects in, 114
See also. Mixed cropping systems (MCS).
- Crop(s)
agricultural, domestication of, 150–152
cover, role in, 191
cultivation of, 681–682
development in fruit, 191
disease resistance in, 45–47
diversity, centers of, 150
domestication of agricultural, 150–152
environment, manipulation of, 703
food, towards residue-reduced, 103–104
and greenhouse management, 545–546
hand harvesting of, 723
insect control
 ant roles in, 109–111
 management complexity, 110
 red imported fire ant in, 113–114
insects, root
 biological control, 132–133
 cultural control, 133
 ecology and control of, 131–134
 insecticidal control, 133–134
interactions in agroecosystems, 712
- [Crop(s)]
and livestock
 non-indigenous species as, 400–402
 risks of low diversity in, 402
losses, 116–120
ornamental
 diseases, 432
 IPM strategy for, 433
 production of, 432
pest tolerance in, 487–488
planting, cultivation after, 338–341
residues
 burning of, 122
 effect, on soil pest populations, 121–122
 need for, 121
 and pest problems, 121–122
 retention of, 121
 role of, 121
rotations
 effects of, 124
 use of, 408
 for weed control, 124–126
protection chemicals, safe
 use of, 588–590
tolerance
 in 21st century, 487–488
 impact of, 488
 non-staple foods, 488
 reduce pesticide, 488
 in staple foods, 487–488
 virus-resistant crops, 45
Cross-resistance to pesticides
cost of, 128–129
future prospects of, 129
mechanisms of, 128
- Cryptolaemus montrouzieri*, 212
- Crysopepla carnea*, 50
- Crystal, protein toxins, 31
- Cry toxins, 553
- Ctenocephalides felis* (cat fleas), 405
- Ctenopharyngodon idella* (grass carp), 34
- Cucumber mosaic virus (CMV), 17
- Cucurbita pepo*, 45
- Culex pipiens*, 350, 715
- Culex quinquefasciatus*, 180, 405
- Culex tarsalis*, 715
life cycle of, 715
- Culicoides furens*, 181
- Curculio caryae* (pecan weevil), 478
role of, 479
- Cybister tripunctatus orientalis*, 40
- Cyclamen mite (*phytonemus pallidus*), 631, 634
- Cyclanilide, 136, 137
- Cyclodienes, 71, 397, 398
- Cydia inopinata* (manchurian codling moth), 474
- Cydia pomonella* (CpGV) (codling moth), 474, 689
- Cydia ptychora* (cowpea seed moth), 106
- Cydia pyrivora* (pear codling moth), 474
- Cylas formicarius*, 333, 502
- Cylas formicarius elegantulu* (sweet potato weevils), 110
- Cylindrocladium black rot (CBR), 470
- Cylindrocladium parasiticum*, 470
- Cynodon dactylon* (bermuda grass), 188, 303
- Cyperus rotundus* (purple nutsedge), 187
- Cyperus esculentus*, 125
- Cytochromes P450, 128, 129, 396
- Cytospora canker (perennial canker), 467
- Dactylopius ceylonicus* (mealybug), 696
- Daktulosphaira vitifoliae* (grape phylloxera), 207
- Dalbulus maidis* (corn leaf hopper), 114
- Danaus plexippus* (monarch butterfly), 255
- Danish groundwater
monitoring program, 218–219
pesticides and metabolites
 found in, 220–221
- Daphnia magna*, 47
- Daphnia pulex*, 40
- D-C-Tron[®], 420
- D-C-Tron Plus[®] (C23), 420
- Deacetylation, 69
- Decision guidance document (DGD), 571
- Deer, red (*cervus elaphus*), 347
- DEET. *See* N,N, diethyl-m-toluamide.
- Defective interfering particles (DIP), 692
- Defoliants
application methods and
 precautions, 136–137
categories of, 135–136
for cotton, 135–137
crop response to, 137
herbicidal, 135, 137
hormonal, 135
- Defoliation, chemical, 135
- Delta-endotoxin (cry), 553
- Delusions of parasitosis. *See* Delusory parasitosis (DP).
- Delusory parasitosis (DP) (delusions of parasitosis, psychogenic parasitosis, Ekbohm's syndrome), 138–140
causes of, 139
delusions of, 138
hallucinogens, effect of, 139
role of entomologists in
 dealing with, 139
symptoms of, 138–139
- Demethylation inhibitors (DMIs), 73
- D-endotoxin, 50
- Dengue
clinical manifestations of, 141–142
diagnosis of, 141
hemostatic abnormalities in, 141
infections in young children, 141
pathogenesis of, 141
phenomenon of, 141
prevention of, 142–143
prognosis of, 141
treatment of, 142
virus, serotypes of, 141
- Dengue fever (DF)
incubation period of, 141
treatment of, 142
- Dengue hemorrhagic fever (DHF), 356
detection of, 141
diagnosis of, 141
hemagglutination–inhibition test (HI test) for, 141
hemostatic abnormalities in, 141

- [Dengue hemorrhagic fever (DHF)]
 laboratory findings in, 141
 pathogenesis of, 141
 treatment of, 142
- Dengue shock syndrome (DSS), 141, 356
 diagnosis of, 141
- Desiccants, 135, 136
- DES. *See* Diethylstilbestrol.
- Desert locust (*schistocerca gregaria*), 320–321
- Designated national authority (DNA), 571
- Deployment trap, 504–505
- Detoxification
 of herbicides, 344
 metabolic, 345
- DGD. *See* Decision guidance document.
- DHF. *See* Dengue hemorrhagic fever.
- Diachasmimorpha longicaudata*, 444
- Diadegma semiclaustum*, 387
- Diamond-back moth (*plutella xylostella*), 132, 242, 335, 387, 553, 554
- Diapetimorpha introita*
 (ichneumonid wasp), 370
- Diaporthe phaseolorum*, 409
- 1,2-Dibromo-3-chloropropane (DBCP), 229, 393
- Dicarboximides, 73
- 2,6-Dichlorobenzamide (BAM), 218, 221
- Dichlorodiphenyltrichloroethane (DDT), 70, 128, 129, 363, 398, 482
- 2,4-Dichlorophenoxyacetic acid (2,4-D), 162
- Dicyphus tamaninii*, 173
- Dieldrin, 71
- Dietary energy supply (DES), 400
- Diethylcarbamazine citrate (DEC), 180
- Diethylstilbestrol (DES), 237, 238
- Diets, artificial, 370–372
 for predators and parasitoids, 370–371
 quality control of natural enemies
 produced on, 371
 successes and failures with, 371
- Dihydropyrazoles, 396, 398
- Dimethipin, 135
- DIP. *See* Defective interfering particles.
- Diplopoda. millipedes, 534
- Dirofilaria immitis*, 352
- Disease resistance
 in crops, 45–47
 methodology for, 14–16
- Diseases
 alfalfa
 control, considerations for, 9, 10
 development of, 7
 ecology and control of, 7–10
 effects of, 7–9
 types of, 8
 crop, ornamental, 432
 dutch elm, 405
 foliar, 647–649
 fungal, 623–624
 agents, 438
 alternaria fruit spot, 437
 anthracnose, 437
 cercospora black spot, 437
 control of, 438
 dry rot, 437
 fruit rot and root rot,
 phytophthora, 437
- [Diseases]
 fusarium fruit rot, 437
 internal blight, 437
 powdery mildew, 437
 fusarium, 645
 glycine max, ecology and control of,
 623–624
 lettuce (*lactuca sativa*)
 causal agents, symptoms, 315–316
 ecology and control of, 313–318
 management of, 315–318
 meleira or sticky, 435
 milky, 30, 658
 mosquito-borne, 357
 monitoring for, 357
 pathogenic forms of, 356
 nematode, 624
 nematode-borne, 436
 ornamental crop, 432, 433
 peach (*prunus persica*)
 brown rot, 465–466
 causal organisms, 466
 ecology and control of, 465–468
 leaf curl, 465
 perennial canker, 467–468
 scab, 468
 peanut (*arachis hypogaea*)
cylandrocladium black rot, 470
 ecology and control of, 469–473
 leaf spots, early and late, 469
 rosette, 470
 rust, 469–470
 stem rot, 470
 phytoplasma, 436
 phytosanitary, 517
 ratoon stunting, 644
 rice (*oryza sativa*), 556
 control options, 560–561
 development of, 560
 ecology and control of, 556–561
 impact of, 556
 survival of pathogen, 557–559
 symptoms of, 557–559
 sclerotinia, 73
 soybean, ecology and control
 of, 623–624
 sugarcane
 bacterial, 644–645
 ecology and control of, 643–645
 fungal, 644–645
 sunflower (*helianthus annuus*)
 ecology and control of, 647–650
 foliar, 647–649
 head rots, 649
 seedling, 647
 stalk and root, 649
 sweet potato, ecology and control of,
 655–656
 tea (*camellia sinensis*)
 ecology and control of, 668–671
 leaf, 669
 management of, 669–671
 of nursery plants, 669
 root, 668
 stem, 668–669
 yellow-type, in papaya, 436
 zoonotic, 181
- Diseases, papaya (*carica papaya*)
 bacterial, 437
 ecology and control of, 435–438
 fungal
 alternaria fruit spot, 437
 anthracnose, 437
 cercospora black spot, 437
 control of, 438
 dry rot, 437
 fruit rot and root rot,
 phytophthora, 437
 fusarium fruit rot, 437
 internal blight, 437
 powdery mildew, 437
 nematode-borne
 reniform, 436
 root knot, 436
 phytoplasma, 436
 viral
 meleira or sticky, 435–436
 mosaic, 435
 ring spot, 435
- Diseases, pea (*pisum sativum*)
 bacterial
 blight, 459
 pink seed, 459
 ecology and control of, 457–459
 fungal, 459
 downy mildew (*peronospora viciae*), 56,
 457
 fusarium wilt (*fusarium oxysporum*), 457
 gray mold (*botrytis cinerea*), 459
 mycosphaerella blight (*mycosphaerella
 pinodes*), 457, 458
 powdery mildew (*erysiphe pisi*), 75, 76,
 437, 457–458
 root rot, aphanomyces and
 fusarium, 458
 seedling blight, 458
 white mold (*sclerotinia sclerotiorum*), 459
 management, strategies for, 459
 nematodes, 459
 viruses, 459
- Disinfestations
 insect and mite, 295–296
 radiation, 296
 soil, 194
- Dispersal
 definition of, 255
 insect pest
 factors affecting, 256–257
 future prospects of, 257
 ways of, 255
- Dithiocarbamates, 73
- Diuron, herbicidal action of, 135
- DNA polymorphism, detection of, 15
- Dolichoderus toracicus*, 109
- Domestication of agricultural
 crops, 150–152
- Domestic ferrets (*mustela vison*), 347
- Domestic pests
 habitats, 246
 infestation of, 245
 overwintering, 245
 prevention and elimination
 strategies for, 245–246
 social, 245

- [Domestic pests]
 solitary, 245
 Dormancy, weed seed, 708–711
 termination of, 709
 Dormant Quick Mix Heavy® oils, 420
 Dormant Soluble®, 420
Doryctobracon toxotrypanae, 444
 Downy brome (*bromus tectorum*), 126
 Downy mildew (*peronospora viciae*), 56, 457
 DP. *See* Delusory parasitosis.
Drosophila melanogaster, 128
Drosophila simulans, 128
Drosophila spp. (vinegar flies), 636
 DSS. *See* Dengue shock syndrome.
 Dusky wireworm (*agriotes obscurus*), 630
 Dutch elm disease, 405
Dysmicoccus morrisoni, 114
- E-17-2 (dihydroxyethylisotridecyloxy propylamine), 1
 EBIs. *See* Ergosterol biosynthesis inhibitors.
Echinochloa glabrescens, 187
 Ecologically based management of rodent pests (EBMRP), concept of, 569
Egeria densa, 639
 EH. *See* Enemies hypothesis.
 Ekbohm's syndrome (delusory parasitosis (DP), delusions of parasitosis, psychogenic parasitosis), 138–140
 causes of, 139
 delusions of, 138
 hallucinogens, effect of, 139
 role of entomologists in dealing with, 139
 symptoms of, 138–139
Elasmopalpus lignosellus (cornstalk borer), 106
 Electromagnetic spectrum, remote sensing of, 594–595
 Elephantiasis, 179, 356
Elodea canadensis, 639
Elymus repens (quackgrass), 304
 Emerald ash borer (*agrilus planipennis*), 654
Empoasca fabae (potato leafhopper), 11
Empoasca insularis, 444
Empoasca papayae, 437, 444
Empoasca stevensi, 437, 444
Empoasca vitis (green grape leafhopper), 387
Encarsia fasciata, 661
Encarsia formosa, 544
Encarsia perniciosi, 477
 Endocrine disruptor chemical (EDC), 237
 Endocytosis, adsorptive, 690
 Entomopoxvirus (EPV), 659
 Endophyte-enhanced grasses, 263, 264
 Endosulfan, 71, 230
 Endotoxin, protein, 31
 Enemies hypothesis (EH), 373
 predictions of, 373
 Enemies, natural, 370–372
 antagonist of, 374
 augmentation of, 299
 bacteria associated with, 455
 biocontrol and artificial diets for rearing, 370–372
 evaluating effectiveness of, 377
- [Enemies, natural]
 fungal pathogens of, 454–455
 future applications, 384
 impact of, 377
 and insecticides, role of, 600
 mass-produced, pathogens of, 453–456
 methods of evaluation
 addition method, 379
 correlation analysis, 378
 exclusion method, 379–380
 interference method, 380
 life tables, 378–379
 microbes, 455
 in mixed cropping systems, functioning of, 373–375
 monitoring postrelease establishment of, 377
 nematodes, 455
 plant food to enhance performance of, 524–526
 protozoan parasites of, 453–454
 quality control
 on artificial diets, 371
 guidelines, 382
 test and methods for production of, 382–384
 viruses and, 455
Enterobacter cloacae, 437
Entomobryoides dissimilis, 429
 Entomopathogenic bacteria, 659–660
 Entomopathogenic fungi, 658
 Entomopathogenic nematodes, 658
 Entomopathogenic viruses, 658
Entypotrachelus meyeri, 672
 Environmental protection agency (EPA), 3, 307
 Enzyme linked immunosorbent assay (ELISA), 323, 451, 492, 690
 Enzyme on paraxon, action of, 69
Ephestia cautella (Mediterranean flour moth), 508
Ephestia kuehniella, 370, 384, 463
Epiphyas postvittana (light brown apple moth (LBAM)), 200, 337
Episyrrhus balteatus, 248
 EPV. *See* Entomopoxvirus.
Eretmocerus mundus, 456
 Ergocalciferol (vitamin D₂) (chronic rodenticides), 568
 Ergosterol biosynthesis inhibitors (EBIs), 73, 215
Erwinia amylovora, 432
Erwinia carotovora, 649
Erwinia chrysanthemi, 655
Erwinia rhapontici, 459
Erysiphe pisi (powdery mildew), 75, 76, 437, 457–458
Erythroneura elegantula, 387
 Ethylene dibromide (EDB), 393
 Ethephon, 136, 137
 Ethylene thiourea (ETU), 218
Etiella zinckenella (pea pod borer), 463
Eucosma notanthes (carambola fruit borer), 502
 Eugregarine spp., 453
 EU-MAC. *See* European Union maximum concentration level.
- Eumorpha achemon* (achemon sphinx moth), 207
Euonymus europaea, 388
Euphyllura olivina (olive psylla), 425
 European agroecosystems
 bioindicator development in, 38–39
 potential bioindicators for sustainability of farming practices in, 39
 European chemicals bureau (ECB), 146
 European corn borer (*ostrinia nubilalis*), 408
 European fly (*compsilura concinnata*), 34
 European Union maximum concentration level (EU-MAC), 220
Euzophera pinguis (olive pyralid moth), 425
Exobasidium vexans, 669
Exorista larvarum, 370
- Facultative predation as biological control, 172–174
 FAO. *See* Food and Agriculture Organization.
 Farmer field school (FFS), IPM approaches, 292–293
 characteristics of, 292
 learning activities in, 293
 principles of, 292
 typical studies of, 293
 Farming practices, sustainability, 37–40
 future studies on bioindicator for, 40
 potential bioindicators for, 39
 use for assessing, 37–40
 Fast-acting or acute rodenticides, 567–568
 fluoroacetamide (1081), 567
 sodium fluoroacetate (1080), 567
 strychnine, 567–568
 zinc phosphide, 568
 FDA. *See* Food and drug administration.
 FDV. *See* Fiji disease virus.
 Federal food, drug, and cosmetic act (FFDCA), 495
 Federal insecticide, fungicide, and rodenticide act (FIFRA), 307, 495
 Fenamidone, 647
 Fenitrothion, 68
 Fenoxaprop, 305
 Fertilizers, nitrogen, 2
Festuca arundinacea, 303
 Few polyhedramutants (FP), 692
 FFS. *See* Farmer field school.
 Fiji disease virus (FDV), 643
 Filarial parasite, physiological strains of, 179
 Filariasis, 179–181
 bancroftian, 179
 brugian, 179, 180
 control of, 180–181
 diagnosis of, 180
 genomes of, 179
 human, 179
 lymphatic, 179
 mosquito-borne, 356
 rural, 179
 symptoms of, 179
 transmission dynamics of, 180
 urban, 179
 vectors for, 180

- Fish and wildlife service (FWS), 404
- Flooding
 cyclical submergence or intermittent, 186
 fallow, 188–189
 physiological adaptations to, 186–187
 systems in rice, 185–186
 weed control, 187
- Fluoroacetamide (1081)
 (acute rodenticides), 567
- Fog
 cold
 advantages of, 164
 aerial spraying, 716, 717
 definition, 164
 technology, 164
 thermal
 advantage of, 164
 applications of, 163–164
- Foliar diseases, 647–649
- Foliar fungicides, 469
- Food and Agriculture Organization (FAO), 279, 552, 570, 581, 583
- Food and Drug Administration (FDA), 358, 531, 532, 720
- Food crops, towards residue-reduced, 103–104
- Food residues, 103
- Food security, genetic diversity for, 519
- Food supplements, use of, 524–526
 artificial, 525
 biological control, 525–526
 mass rearing, 524
- Food webs, 375
- Fopius arisanus*, 444
- Fox (*vulpes vulpes*), 347
- Foxtail (*setaria glauca*), 188
- FP. *See* Few polyhedramutants.
- Fragaria X ananassa*, 630, 631
- Frankliniella occidentalis* (western flower thrips), 214, 631, 636
 biological control of, 546
- Frit fly (*oscinella frit*), 61
- Fruit borer (*leucinodes orbonalis*), 600
- Fruit crop pest management (weeds), 190–193
 agronomic methods
 cover crops, 191–192
 mechanical tillage, 190–191
 mulching, 191
 biological methods, 192–193
 chemical methods, 192–193
 physical methods, 190
- Fruit flies
bactrocera dorsalis, 440
ceratitis capitata (med flies), 652
 papaya, 440, 441
- Fumigants, 568–569, 656
 application methods of, 194–195
 benefits of, 194
 and environment, 194–195
 soil disinfestations, 194
 toxic effect of, 194
 use of, 195
- Fumigation, 194–195, 463
 definition of, 194
 and integrated pest management, 195
 soil, 393
- Fungal diseases, 623–624
 agents, 438
 alternaria fruit spot, 437
 anthracnose, 437
 cercospora black spot, 437
 control of, 438
 dry rot, 437
 fruit rot and root rot, phytophthora, 437
 fusarium fruit rot, 437
 internal blight, 437
 powdery mildew, 437
- Fungal pathogens, 432, 454–455
 control of, 454
- Fungicide(s), 72, 433, 434, 649, 678
 copper, 670
 foliar, 469
 phosphorothiolate, 345
 resistance, feature of, 345
- Fungi, soil-borne, 681
- Fusarium disease, 645
- Fusarium oxysporum* (fusarium wilt), 457, 656
- Fusarium sett rot
 (*fusarium verticillioides*), 645
- Fusarium solani*, 437, 458
- Fusarium subglutinans*, 645
- Fusarium verticillioides*
 (fusarium sett rot), 645
- Fusarium wilt (*fusarium oxysporum*), 457, 656
- GABA. *See* Gamma amino butyric acid.
- GABA-gated chloride channel, 71, 128, 396, 397
- Gaeumannomyces graminis*, 408
- Galandromous occidentalis*, 212
- Galendromous occidentalis*, 634
- Galleria mellonella*, 370
- Gambusia affinis* (mosquito fish), 34
- Gamma amino butyric acid (GABA), 395, 396–397
- GATT. *See* General agreement on tariffs and trades.
- GCPF. *See* Global crop protection federation.
- Gene markers, 14
- General agreement on tariffs and trades (GATT), 654
- Gene transfer (transgenesis), 15, 16
- Genetic diversity for food security, 519
- Genetic engineering in pest control, benefits of, 45
- Genetically modified organisms (GMOs), 166, 167
- Genome maps, animal, 16
- Genotyping techniques, molecular, 16
- Geocoris punctipes*, 371
- Geographical information system (GIS), 299, 594
- Geological survey of Denmark and Greenland (GEUS), 219
- Geotextiles, 700
- GHS. *See* Globally harmonized system.
- Giant toad (*bufo marinus*), 347
- GIS. *See* Geographic information systems.
- Glassy-winged sharpshooter (*homalodisca coagulata*), 207
- GlIOClaDIum trICHODerma*, 434
- Global crop protection federation (GCPF), 499, 581, 583
- Globally harmonized system (GHS), 497
- Global positioning system (GPS), satellite technology, 594
- Global programme of action (GPA), 481
- Glutamate-gated chloride channels, 396–397, 398
- Glyceridic oils (vegetable oils and plant oils) in pest control, 421–422
- Glycine max* (soybean)
 diseases, ecology and control of, 623–624
 fungal pathogens of, 624
- Glyptapanteles militaris*, 390
- GMOs. *See* Genetically modified organisms.
- Gossypium hirsutum*, 135, 595, 620
- GPA. *See* Global programme of action.
- GPS. *See* Global positioning system.
- Gracillariid leaf roller (*caloptilia theivora*), 672
- Granary weevil (*sitophilus granarius*), 44
- Granulovirus (GV), 689
- Grape(s)
 insects
 biological control in California vineyards, 211–212
 ecology and control, 207–214
 of world, 208–212
 IPM, 213
 leaf skeletonizer, 213
 leafroller, 212
 mealybug, 212
 pest
 economic impact of, 207
 management, cultural and physical, 212
 management in California,
 case study, 207
 regulatory control, 212
 production in Australia, 200–206
- Grape phylloxera (*daktulosphaira vitifoliae*), 207
- Grape vine scale (*parthenolecanium persicae*), 200
- Grapholita molesta* (oriental fruit moth), 474
- Grass
 bent, 305
 bermuda, 188, 303
 black, 344
 endophyte-enhanced, 263
 insect-resistant, 263
 Johnson, 187
 Kentucky blue, 303, 304, 306
 New Zealand, 31
 quack, 304
 thatch, 187
 tiger, 187, 643
 turf, 261–263, 268
 insect-resistant, 265
 integrated pest management (IPM)
 program for, 261–263
 management, effective, 263
 pests, 268
 weed management, 303
- Grass carp (*ctenopharyngodon idella*), 34
- Grass grab, New Zealand (*costelytra zealandica*), 31

- GRAV. *See* Groundnut rosette assistor virus.
- Gray mold (*botrytis cinerea*), 215, 459
- Green grape leafhopper (*empeasca vitis*), 387
- Greenhouse management, crop and, 545–546
- Greenhouse plant pathogens
- bacteria, 215–216
 - botrytis cinerea*, 214–215
 - managing, 214–217
 - powdery mildew, 215
 - viruses, 216–217
 - water molds (*pythium* and *phytophthora*), 216
- Green kyllinga (*kyllinga brevifolia*), 303
- Green peach aphid (*myzus persicae*), 345, 435, 463, 635
- Green vegetation index (GVI) image, 595
- Grey squirrel (*sciurus carolinensis*), 347
- Groundnut. *See* Penaut.
- Groundnut rosette assistor virus (GRAV), 470, 471
- Groundnut rosette virus (GRV), 470, 471
- Groundwater
- monitoring program, 218–219
 - chemical, 219
 - pesticides and metabolites found in Danish, 220–221
 - pesticides in, 218–221
- Groundwater monitoring system (GRUMO), 220
- GRV. *See* Groundnut rosette virus.
- GV. *See* Granulovirus.
- GVI. *See* Green vegetation index.
- Gypsy moth (*lymantria dispar*) (LdMNPV), 34, 299, 652, 689
- pest containment program, 653–654
- Habrobracon hebetor*, 43
- Halogenated hydrocarbons (HHC), 393
- Hand-net trap, 247
- HaNPV. *See* *Helicoverpa armigera* nuclear polyhedrosis virus.
- Hantavirus pulmonary syndrome (HPS), 562
- Haplothrips tritici* (wheat trips), 61
- Harmonia axyridis* (Asian ladybird beetle), 245
- Haveli system, 189
- Hawaiian duck (*anas wyvilliana*), 349
- Hazard communication, 223–225
- comprehension of, 225–226
 - effectiveness of, 223
 - evaluation of, 226
 - roles of, 223, 224
 - symbols
 - comprehensibility of, 225
 - for pesticides labels, 223, 224
 - tools, 223
- Hazard labeling, 223–226
- See also* Pesticides label.
- HCH. *See* Hexachlorocyclohexane.
- Helianthus annuus* (sunflower) disease
- ecology and control of, 647–650
 - foliar, 647–649
 - head rots, 649
 - seedling, 647
- [*Helianthus annuus* (sunflower) disease] stalk and root, 649
- Helicoverpa armigera*, 129, 232, 598
- biotechnological approaches, transgenics with
 - Bt*. endotoxin, 235
 - plant proteinase inhibitors, 235
 - control methods, biological
 - bacillus thuringiensis* sprays, 234–235
 - nuclear polyhedrosis virus, 234
 - pheromones, 234–235
 - ecology of pest, 232
 - life cycle of, 234
- Helicoverpa armigera* nuclear polyhedrosis virus (HaNPV), 234
- Helicoverpa zea* (HzSNPV) (corn earworm), 408, 689
- Heliothis* spp., resurgence of, 597
- Heliothis virescens* (tobacco budworm), 114, 128
- Helminthosporium maydis*, 408
- Helminthosporium turcicum*, 408
- Helopeltis theivora*, 672
- Hemagglutination–inhibition test (HI test), 141
- Hemiberlesia cyanophylli*, 672
- Hemileia vastatrix*, 574
- Hemorrhagic fever with renal syndrome (HFRS), 562
- Herbaspirillum rubrisubalbicans*, 644
- Herbicide(s), 72, 706
- acetolactate synthase (ALS)-inhibiting, 170
 - carbamate, 72
 - classification of
 - amino acid biosynthesis inhibitors, 697
 - auxin mimics, 698
 - cell division inhibitors, 697, 698
 - lipid biosynthesis inhibitors, 697
 - photosynthesis inhibitors, 697
 - pigment production inhibitors, 697
 - respiration inhibitors, 698
 - effectiveness of, 407
 - formulation of, 706
 - non-transported, 192
 - phenoxy, 253
 - postemergence, 192, 305, 306, 706
 - preemergence, 305, 706
 - sequestration and detoxification, 169
 - sublethal effects of, 276
 - toxic effects of, 47–49
 - triazine, 170
 - use of, 611, 612
- Herbicide resistance
- fitness costs, 169–171
 - assessment of, 169–170
 - associated with, 169
 - basis for, 169
 - estimation, importance, 169–170
 - related to target-site resistance, 170
 - management of, 170
- Herbicide resistant crops (HRCs)
- technology, 45, 48
 - development of, 51
 - economic impacts of, 49
 - toxicity of, 47
- Herbicide-tolerant grains, 167
- Herpestes auropunctatus* (Indian mongoose), 33, 347
- Hesperomyces virescens*, 454
- Hessian fly (*mayetiola destructor*), 408
- Heterocyclic organophosphates, 68–69
- Hexachlorocyclohexane (HCH, BHC), 70, 252
- isomers of, 71
 - toxic principle in, 71
- Hexythiazox, 300
- Hibiscus sabdariffa*, 444
- High performance liquid chromatography, 6
- HI test. *See* Hemagglutination–inhibition test.
- Homalodisca coagulata* (glassy-winged sharpshooter), 207
- Hormoligosis, 597
- Hormonal disruption
- in men, 238
 - in women, 238
 - mechanisms of chemical, 237
- Hornwort or coontail (*ceratophyllum demersum*), 639
- Host-plant acceptance, 241
- Host plant resistance, 259
- Host-plant selection
- contributing factors, 241–242
 - by insects, 240–243
 - practical considerations, 242
 - theory, 240
- Host specificity of baculoviruses, 691
- Hot Foot[®], 54
- House fly (*musca domestica*), 245
- Household pest management (insects and mites), 244–246
- House mouse (*mus musculus*), 347, 562
- House sparrows (*passer domesticus*), 52, 347
- Hoverflies, 247–250
- collecting methods
 - chromotropic, 247
 - hand-net, 247
 - malaise trap, 247
 - effect of different farming systems on, 250
 - as indicators of biodiversity, 248–249
 - indicators of sustainable farming, 247–250
 - populations, 248
 - in agroecosystems, 248
 - potential control of aphids, 247–250
 - study of, 247
- HPSO[®], 420
- HRCs. *See* Herbicide resistant crops.
- Human filariasis, 179
- Human lymphatic filariasis, 179
- Humans, hormonal disruption in, 237–238
- Hybridization, somatic cell, 492
- Hydrilla verticillata*, 639
- Hydrolysis, Ach, 69
- Hyla japonica*, 40
- Hymenoptera parasitoids, 248
- Hypera postica* (alfalfa weevil), 114
- management of, 11
- Hyperparasitoids, 373

- Hypothenemus hampei* (coffee berry borer), 96
 HzSNPV. *See Helicoverpa zea*.
- IBMA. *See* International Biocontrol Manufacturers Association.
- Ichneumonid wasp (*diapetimorpha introita*), 370
- ICRC. *See* Interim chemical review committee.
- ICT. *See* Immunochromatographic test.
- IFOAM. *See* International Foundation for Organic Agriculture Movements.
- Imidacloprid, 300
 efficacy of, 65
 formulation, effects of, 64
 irrigation of, 65
- Immune deficiency effects, in laboratory animals, 251
- Immunochromatographic test (ICT), 180
- Immunotoxic effects in man, 251–254
 carbamates, 252
 organochlorines insecticides, 251
 organophosphorous compounds, 251
 organotin compounds, 253
 pentachlorophenol (PCP), 253
 phenoxy herbicides, 253
- Immunotoxicity, 251
 of pentachlorophenol (PCP), 253
- Impatiens necrotic spot virus (INSV), 214, 216
- Imperata cylindrica* (thatch grass), 187
- INC. *See* Intergovernmental negotiating committee.
- Indarbela theivora*, 672
- Indian meal moth (*plodia interpunctella*), 245, 507, 553
- Indian mongoose (*herpestes auropunctatus*), 33, 347
- Ind-Ne[®], 422
- Infochemicals, plant-derived, 374
- Information exchange network on capacity building for sound management of chemicals (INFOCAP), 146
- Inhibitors
 amino acid biosynthesis, 697
 auxin transport, 136
 cell division, 697, 698
 demethylation, 73
 ergosterol biosynthesis, 73, 215
 in vitro, 69
 lipid biosynthesis, 697
 photosynthesis, 697
 photosystem II (PSII), 170
 pigment production, 697
 plant proteinase, 235
 reproductive, 54
 respiration, 698
 sterol, 77
- Insect(s)
 alfalfa, 11–12
 ecology and management of, 11–12
 black scale, 427
 captive rearing of, 175–178
 for field release, 175–178
 paradox of, 175
- [Insect(s)]
 cherry, ecology and control of, 79–86
 coconut, ecology and control of, 90–91
 coffee, ecology and control of, 95
 control
 ant roles in crop, 109–111
 fire ant in crop, red
 imported, 113–114
 management complexity, 110
 cowpea
 ecology and control of, 106–108
 pests, 106–107
 cruciferous root crop
 ecology and control of, 131–134
 biological control, 132–133
 cultural control, 133
 insecticidal control, 133–134
 entomophagous, culture of, 370
 grape
 biological control in California vineyards, 211–212
 ecology and control, 207–214
 of world, 208–212
 invasion phenomenon, 288
 by sea or by air, 289–290
 invasive, 288–289
 management systems, implementation of, 260
 and mite
 disinfestations, 295–296
 pests and control tactics, 544–546
 monitoring techniques, 261–263
 olive, ecology and control of, 425–427
 papaya
 ecology and control of, 440–445
 sampling and monitoring of, 440
 pear
 biological control, 477
 ecology of, 474–477
 pest control, insecticidal, 474–476
 pecan
 control, 479
 ecology, 478–479
 pest
 causes of, 597
 factors influencing pesticide-induced, 598
 pest dispersal
 definition of, 255
 factors affecting, 256–257
 future prospects, 257
 migration, types, 255–256
 ways of, 255
 pest management, 258–260, 600
 for lawns, 261–266
 technologies and procedures, 259–260
 pheromones, applications of, 502
 and plant pathogens, release of compounds toxic to, 428
 populations, dispersal of, 255
 root crop
 biological control, 132–133
 cultural control, 133
 ecology and control of, 131–134
 insecticidal control, 133–134
 social, 245
 solitary, 245
 surface-feeding, 122
- [Insect(s)]
 vectors, chemical control, 561
 virus
 associated to, 689
 for biological control of, 689–692
 characterization of, 690–691
 identification of, 690–691
 Insect growth regulators (IGRs), 85, 129
 development of, 546
 Insecticidal cultivars, transgenic, 553
 Insecticide(s), 67, 70, 265, 398, 717
 abamectin (neonicotynyl), applications of, 475
 actions of, 395
 aerial application of, 717
 for adult mosquito, 717
 classes, neurotoxic, 398
 avermectin, 398
 carbamate, 398
 chloronicotynyl, 398
 cyclodiene, 398
 DDT and analogs, 398
 dihydropyrazole, 398
 milbemycin, 398
 neonicotinoid, 398
 organophosphate, 398
 phenylpyrazole, 398
 polychlorocycloalkane, 398
 pyrethrin, 398
 pyrethroid, 398
 spinosoid, 398
 spinosyn, 398
 effectiveness, in reaching target mosquitoes, 717
 examples of, 398
 foliar-applied contact, 666
 natural enemies and, role of, 600
 neurological effects of, 395–399
 neurotoxic, 395
 classes, 398
 target sites and effects of, 398
 organochlorines, 251
 organophosphate, 395
 organophosphorus
 biological activity of, 68
 chemical classes of, 68–69
 pyrethroid, 67
 reduction on lawns, 267–269
 biological control, 268
 chemical control, 269
 fertilization, 267
 irrigation, 267
 mechanical control, 268
 residue monitoring, 5–6
 resistance in colorado potato beetle, 272
 resistance management, 271–273
 monitoring, 272–273
 practical considerations, 272–273
 soil-applied systemic, 665, 666, 667
 systemic
 benefits of, 665–666
 characteristics of, 664–665
 costs of, 666–667
 target sites, neurological, 395–399
 use to minimize resistance, 271–272
 Insecticide-treated bednets (ITNs), 358

- Insectistasis, 509
- Insect-resistant grasses, 263
- INSV. *See* *Impatiens necrotic spot virus*.
- Integrated disease management, examples of, 58
- Integrated pest management (IPM), 47, 63–64, 145, 207, 279, 298, 511
- aims of, 280
 - in alfalfa, 11
 - components of, 279
 - concept of, 280, 507
 - cornerstones of, 544
 - crucial factors for, 509
 - definitions of, 279
 - development of, 173, 507
 - ecological aspects of, 281
 - essential strategy of, 280
 - implementation of, 73, 172, 686
 - for ornamental diseases, 432
 - for ornamentals, examples, 432–434
 - pheromones in, 507–510
 - principles of, 275–278
 - programs, 428
 - recommendations, risks of plant feeding–incorporation, 172–173
 - role of, 592
 - systems, development of, 544
 - tactics of, 280
 - use of, 686, 687
- Integrated pest management (IPM) program, 583
- disadvantage of, 583
 - for turfgrass, 261–263
 - detection, early, 261
 - insect monitoring techniques, 261–263
 - pest identification, 261
 - record keeping, 263
- Integrated plant control, system and management of, 281
- definition and history, 279–280
 - diagnosis, 280–281
 - prognosis and signalization, 281
- Integrated weed management, 303
- Inter-governmental forum on chemical safety (IFCS), 146
- Intergovernmental negotiating committee (INC), 571
- Interim chemical review committee (ICRC), 571
- Internally transcribed spacer (ITS), 661
- International Biocontrol Manufacturers Association (IBMA), 382
- International Foundation for Organic Agriculture Movements (IFOAM), 60
- International Organisation for Biological and Integrated Control of Noxious Animals and Plants (IOBC-OILB), definition of, 279
- International Organization for Biological Control (IOBC), 382
- International pesticide poisoning surveillance, 283–287
- International Programme on Chemical Safety (IPCS), 283, 285
- International Rice Research Institute (IRRI), 386, 519
- Inter-organization programme for sound management of chemicals (IOMC), 146, 225
- Invasive insects
- invasion phenomenon, 289
 - as major pests in United States, 288–290
- Invasive species, terrestrial
- characteristics of, 347
 - economical and ecological impacts of, 348
 - management of, 349
 - mitigating impacts of, 347–349
- IOBC. *See* International Organisation for Biological Control.
- IOBC-OILB. *See* International Organisation for Biological and Integrated Control of Noxious Animals and Plants.
- Iowa survey, 727
- IPM. *See* Integrated pest management.
- IPM farmer field school (FFS)
- approaches, 292–293
 - characteristics of, 292
 - learning activities in, 293
 - principles of, 292
 - typical studies of, 293
- Ipomoea batatas* (sweet potato), 655–656
- Iprodione, 77
- Irradiation, 295–297
- of foods, 295
- IRRI. *See* International Rice Research Institute.
- Ivermectin, 180, 181, 398
- Japanese beetle (*popillia japonica*), 30
- Jasmine moth (*palpita unionalis*), 425
- Johnson grass (*sorghum halepense*), 187
- Juvenile hormone analogues (JHA), 416
- Keiferia lycopersicella* (tomato pinworm), 336
- Kentucky bluegrass (*poa pratensis*) turf, 303, 304, 306
- Krebs cycle (tricarboxylic acid cycle), 567
- Kyllinga brevifolia* (green kyllinga), 303
- Labels, pesticides, 223–226, 495–497
- attributes of
 - environment, 224–225
 - message, 223–224
 - reader, 224
 - components
 - colors, 223
 - signal words, 223
 - symbols, 223
 - development of, 495–497
 - global harmonization of, 225–226
 - initiatives, 497
 - types of
 - primary, 497
 - special local need (SLN) label, 497
 - supplemental distributor, 497
- Lactuca sativa* (lettuce)
- crisphead, 313
 - growth stages of, 314
- [*Lactuca sativa* (lettuce)]
- diseases
 - causal agents, symptoms, 315–316
 - ecology and control of, 313–318
 - management of, 315–318
 - production of, 313
 - types of, 313
- Lactuca serriola*, 313
- Lampides boeticus*, 463
- Landscape ornamentals, 298–301
- biological diversity of, 298–299
 - decision making in, 299
 - intervention tactics and strategies, 299–300
 - biological control, 299–300
 - chemical controls, 300
 - cultural tactics, 300
 - host plant resistance, 300
 - new approaches for monitoring, 299
 - rationale for pest management in, 298
- Lantana camera*, 188, 696
- Lantbrukarnas Riksförbund (LRF), 585
- Lantern trap, 503
- Lariophagus distinguendus*, 42
- Lasiodiplodia theobromae*, 656
- Lasius neoniger*, 109
- Lathocerus medius*, 537
- Latrodectus hesperus*, 532
- Latrodectus mactans*, 532
- Latrodectus*, toxins of, 532
- Latrotoxins, 532
- Lawn-care treatment (weeds), 303–306
- chemical, 304–306
 - cultural, 303–305
 - integrated weed management, 303
 - prevention, 303
- Lawns
- insecticide reduction on, 267–269
 - insect pest management for, 261–266
- LBAM. *See* Light brown apple moth.
- LdMNPV. *See* *Lymantria dispar*.
- Leafhopper, corn (*dalbulus maidis*), 114
- Leafhopper, green grape (*emposasca vitis*), 387
- Leafhopper, potato (*emposasca fabae*), 11
- Leafminers, 461–462
- Leggett trap, 503
- Legume pod borer (*maruca vitrata*), 106
- Leifsonia xyli*, 644
- Lema beetles (*oulema lichenis*)
- economic importance of, 446
 - as host of parasites, 446, 447
 - occurrence of, 447
 - parasites on, 446–448
 - spectrum of, 447
- Lema melanopus*, 446, 447
- Lemophagus curtus*, 448
- Leopard moth (*zeuzera pyrina*), 425
- Lepidoptera, 635
- Leptinotarsa decemlineata* (colorado potato beetle (CPB)), 665
- biological cycle, seasonal, 100
 - control of, 100
 - dispersal of, 256
 - impact of heat on, 99
 - insecticide resistance in, 272
 - mortality rates of, 100

- [*Leptinotarsa decemlineata* (colorado potato beetle (CPB))]
 thermal control of, 99–101
 efficacy of, 99–100
 visual index damage, 99
- Leptochloa chinensis*, 187
- Lethal dose (LD₅₀), 723, 691
- Lethal stressing agents, 53
- Lethal synthesis, 567
- Lethal time (LT₅₀), 691
- Lethal toxicants, 52, 53
- Lethocerus americanus*, 536
- Lethocerus deyrollei*, 40
- Lettuce (*Lactuca sativa*)
 crisphead, 313
 growth stages of, 314
 diseases
 causal agents, symptoms, 315–316
 ecology and control of, 313–318
 management of, 315–318
 production of, 313
 types of, 313
- Lettuce mosaic virus (LMV), 317
- Leucinodes orbonalis* (fruit borer), 600
- Leucoptera coffeella* (coffee leaf miner), 95–96
- Leucostoma cinctum*, 467
- Leucostoma persoonii*, 467
- Leukosis, bovine, 14
- Levuana iridescens*, 93
- Light brown apple moth (LBAM)
 (*epiphyas postvittana*), 200, 337
- Limothrips denticornis* (rye thrips), 61
- Liothrips oleae* (olive thrips), 425
- Lipaphis erysimi* (mustard aphid), 598
- Lipid biosynthesis inhibitors, 697
- Liriomyza huidobrensis*
 (pea leafminer), 390, 461
- Liriomyza sativae* (vegetable leafminer), 462
- Liriomyza trifolii* (American serpentine leafminer), 462
- Litchi fruit borer (*conopomorpha sinensis*), 504
- Livestock and crops
 non-indigenous species as, 400–402
 risks of low diversity in, 402
- Living modified organisms (LMOs),
 development of, 363
- LMV. *See* Lettuce mosaic virus.
- Loa loa*, 179
- Locust control, by early identification of
 breeding sites, 319–321
- Locusta migratoria migratorioides* (African migratory locust), 319
- Locustina pardalina* (brown locust), 320
- Loiasis, 181
- Lolium multiflorum*, 388
- Long-range transboundary air pollution
 (LRTAP), 481
- Long-range transportable air pollutants
 (LRTAPs), 412
- Long-tailed mealybug
 (*pseudococcus longispinus*), 200
- Loxistege sticticalis* (beet webworm), 461
- Loxosceles arizonica*, 532
- Loxosceles deserta*, 532
- Loxosceles intermedia*, 532
- Loxosceles laeta*, 533
- Loxosceles reclusa*, 532
- LRTAP. *See* Long-range transboundary air pollution.
- LRTAPs. *See* Long-range transportable air pollutants.
- Ludiwigia octovalvis*, 187, 188
- Lycopersicon esculentum*, 595
- Lygus bug
 and forage alfalfa, 322
 management by alfalfa harvest
 manipulation, 322–324
 within-field movement of, 323
- Lygus hesperus* (western tarnished plant bug), 106, 631, 635
- Lygus linoelaris* (tarnished plant bug),
 463, 631
- Lymantria dispar* (LdMNPV) (gypsy moth),
 34, 299, 652, 653–654, 689
- Lymphatic filariasis, 179, 350
- Lymphomas, non-Hodgkin's,
 development of, 724
- Maconellicoccus hirsutus*, 405
- Macrolophus caliginosus*, 173
- Macrophomina phaseolina*, 656
- Maculina arion* (blue butterfly), 35
- Major histocompatibility complex
 (MHC), 14
 genes, role, 15
 phenotype, classes of, 14
- Malaise trap, 247
- Malathion, 69
- Mammal pest
 control of, 326
 impacts in New Zealand, 329–330
- Mammal traps
 control programs, efficiency and
 costs, 326
 efficiency of, 326
 future needs of, 327
 role in, 327
 strategies and concerns, 327
 types of, 326
- Manchurian codling moth
 (*cydia inopinata*), 474
- Man, immunotoxic effects in, 251–253
- carbamates, 252
- organochlorines insecticides, 251
- organophosphorous compounds, 251
- organotin compounds, 253
- pentachlorophenol (PCP), 253
- phenoxy herbicides, 253
- Mansonella ozzardi*, 179
- Mansonella streptostraca*, 179
- Mansonella streptostraca*, 179
- Mansonellosis, 179, 181
- Maps, animal genome, 16
- MARD. *See* Ministry of Agriculture and Rural Development.
- Marker-assisted selection (MAS), 14
- Markers, gene, 14
- Marsilea minuta*, 187
- Maruca vitrata* (legume pod borer), 106
- Mass-produced natural enemies,
 pathogens of, 453–456
- Mass-trapping
 attractant source, 332
 experiments on pheromone-based, 333
 lure-and-kill-type formulations, 332
 practical use of pheromone-based, 333
 using pheromones and
 semiochemicals, 334
- Mastitis, 14
- Material safety data sheets (MSDSs), 2
- Mating disruption, 336–337
- Mattesia trogodermae*, 335
- Maximum residue limit (MRL), 689
- Mayetiola destructor* (hessian fly), 408
- MCS. *See* Mixed cropping systems.
- MDR. *See* Multidrug resistance.
- Meadow voles, 694
- Mealybug, citrus (*planococcus citri*), 504
- Mealybug (*dactylopius ceylonicus*), 696
- Mealybug, long-tailed (*pseudococcus longispinus*), 200
- MEAs. *See* Multilateral environmental agreements.
- Medflies (*ceratitis capitata*), 440
- Medicago sativa* (alfalfa)
 configuration of uncut, 323–324
 diseases
 control, considerations for, 9, 10
 development of, 7
 ecology and control of, 7–10
 effects of, 7–9
 types of, 8
 harvest manipulation, 322–324
 insects, ecology and
 management of, 11–12
 integrated pest management in, 11
- Mediterranean flour moth
 (*ephestia cautella*), 508
- Melanagromyza aeneoventris*, 390
- Melanagromyza sojae*
 (soybean stem miner), 462
- Melanaphis sacchari*, 644
- Melanaspis glomerata*, 600
- Melanoplus spretus* (rocky mountain locust), 320
- Melanous okinawensis* (sugarcane wireworms), 333
- Meleira or sticky diseases, 435
- Melia azedarach* (chinaberry), 416
- Meloidogine arenaria*, 436
- Meloidogine hapla*, 436
- Meloidogine incognita*, 436
- Meloidogyne incognita*, 361, 436, 595
- Melon aphids (*aphis gossypii*), 114, 248,
 435, 595, 635
- Melon flies (*bactrocera cucurbitae*), 440
- Men, hormonal disruption in, 238
- Meristem tip culture, 492
- Mesuro® (Methiocarb), 54
- Metabolic activation of pesticides, 723
- Metabolic detoxification, 345
- Metabolic resistance, features of, 344
- Metabolism, anaerobic, 187
- Metaphycus hageni*, 427
- Metaphycus helvolus*, 427
- Metarhizium anisopliae*, 268, 454
- Metaseiulus occidentalis*, 453
- Meteorius communis*, 390

- Methamphetamines, 139
 Methiocarb (Mesuroi[®]), 54
 Methoxyfenozide, 85
 Methyl anthranilate, 54
 Methyl bromide, 85, 194, 393
 Methyl isothiocyanate (MIT) liberator, 393
 Methyl parathion, 68
 Michigan survey, 727–728
 Micrografting, 492
 Microorganisms
 antagonistic
 activity of, 428
 stimulation of, 428
 control of, 296–297
 synergy with, 658–660
 Microsatellites, 199
 Microsporidia, 453
 Microsporidian spores, 453
Microtonus hyperodae, 387
 Migration, types of, 255–256
 Milbemycin, 398
Milesia crabroniformis, 250
 Milky disease, 30, 658
Mimosa invisa, 670
 Ministry of Agriculture and Rural
 Development (MARD), 593
Misgurnus anguillicaudatus, 40
 Mite(s)
 broad, 444
 in California, ecology and
 control, 634–636
 cyclamen, 631, 634
 and insect disinfestations, 295–296
 papaya leaf edge roller, 444
 red, 598
 spider, 631–632
 MIT. *See* Methyl isothiocyanate liberator.
 Mixed cropping systems (MCS), 373
 characteristics of, 373
 effects of, 373
 function in, 372–375
 indirect interaction in food webs, 375
 monoculture and, 375
 natural enemies to, 375
 and resource subsidies, 373
 vegetation structure, 374
 volatile information, 374–375
 Molecular genotyping techniques, 15
 Monarch butterfly (*danaus plexippus*), 255
 Monazoline-O (oleyl imidazoline), 1
Monellia caryella (pecan aphid), 478
 Mongoose, Indian (*herpestes
 auropunctatus*), 33, 347
Monilinia fructicola, 466
Monilochaetes infuscans
 (scurf pathogen), 655
 Monitoring
 of animal pest, 61
 in spring barley, 61
 in winter wheat, 61–62
 approaches for, landscape
 ornamentals, 299
 insect residue, 5–6
 of mosquitoes, 357
 for mosquito-borne diseases, 357
 for natural enemies, 377–381
 of olive fly, 425
 [Monitoring]
 of papaya insects, 440
 programs in Europe and USA, 218
 techniques, insect, 261–263
 for voles, 693–694
 Monk parakeets (*myiopsitta monachus*), 347
Monochamus alternatus, 503
Monochamus leuconotus, 96
Monocrepidius vespertinus, 408
 Monocrotophos, applications, 597
 Monogenic resistance, 520, 522
Monolinia fructicola, 75
 Montmorency (tart cherry), 75, 79
 See also Cherry.
 symptoms on, 76
 Mosquitoes
 biology and behavior of, 356
 classification and recognition of, 350
 control of, 353–355, 357–358, 715–719
 aerial ultra low volume (ULV),
 applications, 4–6, 717
 bird reservoirs, 715
 insecticide effectiveness in reaching
 target, 717
 larva, 715
 with predators, 716
 spray technology, 5
 ultralow volume spraying, 716–717
 vectors, 715
 equipment for ground applications
 to, 163–165
 thermal fog, 163–164
 ULV (cold fog), 164
 host preference, 351
 larval control of, 715
 life cycle of, 350–351
 monitoring of, 357
 repellents, 358
 vectors, 715
 Mosquito-borne diseases, 351, 356
 monitoring for, 357
 pathogenic forms of, 356
 Mosquito-borne filariasis, 356
 Mosquito fish (*gambusia affinis*), 34
 Mouse
 house (*mus musculus*), 347, 562
 spiny (*acomys coharinus*), 562
 Movement protein (MP) genes, 435
 MRL. *See* Maximum residue limit.
 Mulches
 advantages of, 191, 362
 economical and technical considerations,
 362
 effects of, 360
 examples of, 360–362
 in vegetables, 361
 in orchards, 361–362
 and pests, 360–362
 system, complexity of, 360
 types of, 12
 use of artificial or organic, 360
 Multidrug resistance (MDR), 345
 Multilateral environmental agreements
 (MEAs), 363
 basel convention, 364
 biodiversity convention and cartagena
 protocol, 364–365
 [Multilateral environmental agreements
 (MEAs)]
 in chemical safety and protection of
 biodiversity, 364
 environmental law, development, 363–364
 pest management and environment, 363
 rotterdam convention, 365
 stockholm convention, 365
 and UN-ECE POP protocol, 365
 Multimammate rat (*mastomys* or *praomys
 natalensis*), 562
Musca domestica (house fly), 245
Mus musculus (house mouse), 347, 562
 Muscarinic receptors, 398
 Muskrat (*ondatra zibethicus*), 347
 Mustard aphid (*lipaphis erysimi*), 598
Mustela vison (domestic ferrets), 347
Mycosphaerella arachidis, 469
Mycosphaerella blight (ascochyta blight
 complex), 457
 life cycle of, 458
Mycosphaerella fijiensis, 573
Mycosphaerella pinodes (mycosphaerella
 blight), 457
 life cycle of, 458
Myiopsitta monachus (monk parakeets), 347
Myocastor coypus, 347
Myrmica sabuleti, 35
Myrothecium verrucaria, 394, 436
 Myxoma virus, 35
Myzus persicae (green peach aphid), 345,
 435, 463, 635

Nasonia vitripennis, 456
 National Agricultural Pest Information
 System (NAPIS), 653
 National pesticide poisoning surveillance,
 367–369
 National Plant Germplasm Quarantine
 Center (NPGQC), 517, 518
 National Water-Quality Assessment
 (NAWQA) program, 610
 Natural enemies, 370–372
 antagonist of, 374
 augmentation of, 299
 bacteria associated with, 455
 biocontrol and artificial diets for
 rearing, 370–372
 evaluating effectiveness of, 377
 fungal pathogens of, 454–455
 future applications, 384
 impact of, 377
 and insecticides, role of, 600
 mass-produced, pathogens of, 453–456
 methods of evaluation
 addition method, 379
 correlation analysis, 378
 exclusion method, 379–380
 interference method, 380
 life tables, 378–379
 microbes, 455
 in mixed cropping systems,
 functioning of, 373–375
 monitoring postrelease
 establishment of, 377
 nematodes, 455

- [Natural enemies]
 plant food to enhance
 performance of, 524–526
 protozoan parasites of, 453–454
 quality control
 on artificial diets, 371
 guidelines, 382
 test and methods for production of,
 382–384
 viruses and, 455
- Natural vegetation, 387–390
 ecological functions of, 387–389
 management to improve parasitoids in
 farming systems, 387–390
 on parasitoids, 387–389
- NAWQA. *See* National Water-Quality
 Assessment Program.
- Necremnus leucarthros*, 447
- Necrosis
 papaya apical, 436
 papaya droopy, 436
- Nectar and pollen feeding, impact of, 524
- Neemark[®], 422
- Neem oil, 416, 417
- Nematicides, 392, 610
 lack of development of, reasons for, 394
 types of, 392
 fumigant, 393
 non-fumigant, 393
- Nematocera (diptera) larvae, 31
- Nematode(s)
 diseases, 624
 entomopathogenic, 658
 life cycle of, 392
 management of, 392
 parasites, 179–181
 plant-parasitic, 392
 reniform, 436, 656
 root knot, 436, 656
- Nematode-borne diseases, 436
- Neoclytus cacticus*, 97
- Neodiprion sertifer* (NeseNPV), 689
- Neonicotinoids, 72, 398, 399
- Neonicotynl (abamectin) insecticides,
 applications of, 474
- Neoplectana glaseri*, 453
- Neoseiulus citrifolius*, 454
- Neoseiulus cucumeris*, 453, 631
- Nephotettix virescens*, 560
- NEPSC. *See* Northeast Exotic Pest Survey
 Committee.
- Nervous system function and
 terminology, 395
- NeseNPV. *See* *Neodiprion sertifer*.
- Nesidiocoris tenuis*, 173
- Net energy of lactation (NEL), 324
- Neuroexcitation, 397, 399
 effects of, 395
- Neuroinhibition, 395, 397
- Neurological effects of insecticides, 395–399
- Neurological insecticide target
 sites, 395–399
- Neurotoxicants, 395
- Neurotoxic insecticides, 395
 classes, 398
 avermectin, 398
 carbamate, 398
- [Neurotoxic insecticides]
 chloronicotinyl, 398
 cyclodiene, 398
 DDT and analogs, 398
 dihydropyrazole, 398
 milbemycin, 398
 neonicotinoid, 398
 organophosphate, 398
 phenylpyrazole, 398
 polychlorocycloalkane, 398
 pyrethrin, 398
 pyrethroid, 398
 spinosoid, 398
 spinosyn, 398
 target sites and effects of, 398
- Neurotoxic pesticides, 396
- Neurotoxins, 531
- Neurotransmitters, 395
 excitatory, 399
- New world screwworm (NWS), 484, 485, 486
- New Zealand, grass grab (*costelytra
 zealandica*), 31
- New Zealand, mammal pest
 impacts in, 329–330
- Nezara viridula*, 106
- Nicotinic acetylcholine receptor, 398–399
- Nilaparvata lugens* (brown plant hopper),
 293, 539, 552, 597
- Nimbecidine, application of, 418
- Nixalite (porcupine wire), 512
- Nitrogen, fertilizers, 2
- N,N, diethyl-m-toluamide (DEET)
 (synthetic repellents), 358, 717
 concentrations of, 353
 formulations of, 353
- No-exit trap, 504
- Nomadacris septemfasciata* (red locust), 319
- Non-Hodgkin's lymphomas,
 development of, 724
- Non-indigenous species
 crops and livestock, 400–402
 risks of low diversity in, 402
- pests, 404–406
 dwellings and vehicles, 406
 forests, 405
 glasshouse crops, 405
 honey bees, 405–406
 human health, 405
 livestock health, 405
 natural environments, 405
 pet animals, 405
 rangeland, pastures, turf and gardens
 stored products, 406
- Non-sticky traps, 504
- Northeast Exotic Pest Survey Committee
 (NEPSC), 653
- Norway rat (*rattus norvegicus*), 562
- No-till on disease problems,
 effects of, 408–409
- No-till on insect problems, effects of, 407
- No-till on weed management, effects of, 408
- No-till system, 407
- Nozzle
 and considerations, 410
 droplet size spectra, 410
 types
 deflector/flooding, 410
- [Nozzle]
 pre-orifice, 410
 single-elliptical-orifice, 410
 venturi/air-induction, 410
- NPGQC. *See* National Plant Germplasm
 Quarantine Center.
- NPV. *See* Nucleopolyhedroviruses.
- Nuclear stock selection, 490
- Nucleopolyhedroviruses (NPV), 658, 659, 689
- NWS. *See* New world screwworm.
- Obsolete pesticides
 guidelines related to, 415
 management of, 412
 reasons for accumulation of, 413
 stocks, 413
 types of, 414
- Occluded virus (OV), 689
- Occlusion-derived viruses (ODV), 690
- Octylphenol (OP), 2
- Octylphenol polyethoxelate (OPP), 2
- ODV. *See* Occlusion-derived viruses.
- OECD. *See* Organization for Economic
 Cooperation and Development.
- Oecophylla longinoda*, 109
- Oecophylla smaragdina*, 109
- Oidium caricae*, 437
- Oidium neolycopersici*, 215
- Oils
 characteristics of, 416–417
 in disease control, 423
 efficacy of, 417
 features of winter and summer
 management of pests with, 417
 glyceridic, 421–422
 for pest control, 417, 420, 422
 phytotoxicity of, 417
 unsulfonated residue (UR) in, 417
 in winter and summer management
 of pests, 417–420
- Oleander scale (*aspidiotus nerii*), 425
- Olive bark beetle (*phloeotribus
 scarabaeoides*), 425
- Olive fly (*bactrocera oleae*)
 life history of, 425
 management of, 425–426
 monitoring of, 425
- Olive insects, ecology and control
 of, 425–427
- Olive moth (*prays oleae*), 425, 426
- Olive psylla (*euphyllura olivina*), 425
- Olive pyralid moth (*euzophera pinguis*), 425
- Olive scale (*parlatoria oleae*), 425
- Olive thrips (*liothrips oleae*), 425
- Omnivory, 172
- Onchocerca volvulus*, 179
- Ondatra zibethicus* (muskrat), 347
- OpMNPV. *See* *Orgyia pseudotsugata*.
- OPs. *See* Organophosphates.
- Opuntia spinosissima* (semaphore cactus), 34
- Orchards, 361–362
 vole management for, 693–695
- Orchex 796[®], 420
- Organic soil amendments, 428–430
 effect of, 430
 efficacy of, 429

- [Organic soil amendments]
 impacts of, 428
 mechanisms of action, 429
 and plant resistance, 428
 use of, 429
- Organization for Economic Cooperation and Development (OECD), 144, 363
 member states, 144–145
- Organochlorines (OCs), 67, 237, 588
 insecticides, immunotoxic effects
 in man, 251
 metabolites of, 723
- Organohalogens, 237
- Organophosphates (OPs), 67, 398, 588, 589, 597
 aliphatic, 68
 application of, 85
 and ChE enzyme, reaction
 between, 69–70
 heterocyclic, 68–69
 phenyl, 68
- Organophosphorus insecticides, 228
 biological activity of, 68
 chemical classes of, 68–69
- Organosilicone surfactants, 1
- Organotin compounds, 73
 immunotoxic effects in man, 253
- Orgyia pseudotsugata* (OpMNPV), 689
- Oriental fruit fly (*dacus dorsalis*), 333
- Oriental fruit moth (*grapholita molesta*), 474
- Orius laevigatus*, 371, 631
- Orius tristicolor* (pirate bug), 621
- Ornamental crop
 diseases, 432
 IPM strategy for, 433
 production of, 432
- Ornamental diseases, control of, 433
- Ornamentals, landscape, 298–301
 biological diversity of, 298–299
 decision making in, 299
 intervention tactics and
 strategies, 299–300
 biological control, 299–300
 chemical controls, 300
 cultural tactics, 300
 host plant resistance, 300
 new approaches for monitoring, 299
 rationale for pest management in, 298
- Ornamental nurseries, weed management
 in, 705–707
- Ornitrol® (Azocosterol), 54
- Oryctes rhinoceros* (coconut rhinoceros beetle), 92, 93
- Oryctolagus cuniculus*, 347
- Oryza nivara*, 187
- Oryza sativa* (rice) diseases, 556
 control options
 chemical, 560–561
 cultural practices, 560
 resistant varieties, 560
 development of, 560
 ecology and control of, 556–561
 impact of, 556
 survival of pathogen, 557–559
 symptoms of, 557–559
- Oryzias latipes*, 40
- Oscinella frit* (frit fly), 61
- Ostrinia nubilalis* (European corn borer),
 50, 384, 408, 553
- Otiorynchus ovatus* (strawberry root weevil), 631
- Otiorynchus sulcatus* (black vine weevil), 631
- Oulema gallaeciana*, 447
- Oulema lichenis* (lema beetles)
 economic importance of, 446
 as host of parasites, 446, 447
 occurrence of, 447
 parasites on, 446–448
 spectrum of, 447
- OV. *See* Occluded virus.
- Oxathiins, 73
- Oxidative stress, development of, 723
- Oxyethylene adjuvants, 2
- PA-14 (Tergitol®), 53
- Paecilomyces fumosoroseus*, 454
- Paecilomyces lilacinus*, 428
- Paenibacillus lentimorbus*, 30
- Paenibacillus (bacillus) popilliae*, 30
 culture and control of, 30
- Palpita unionalis* (jasmine moth), 425
- Panonychus ulmi* (red mite), 598
- Pantala flavescens*, 40
- Papaya (*carica papaya*)
 apical necrosis, 436
 arthropods associated to, 441–444
 droopy necrosis, 436
 fruit flies, 440, 441
 insects
 ecology and control of, 440–445
 sampling and monitoring of, 440
 yellow-type diseases in, 436
- Papaya (*carica papaya*) diseases
 bacterial, 437
 ecology and control of, 435–438
 fungal
 alternaria fruit spot, 437
 anthracnose, 437
 cercospora black spot, 437
 control of, 438
 dry rot, 437
 fruit rot and root rot,
 phytophthora, 437
 fusarium fruit rot, 437
 internal blight, 437
 powdery mildew, 437
- nematode-borne
 reniform, 436
 root knot, 436
- phytoplasma, 436
- viral
 meleira or sticky, 435–436
 mosaic, 435
 ring spot, 435
- Papaya leaf edge roller mite
 (*calacarus brionese*), 444
- Papaya meleira virus (PmeV), 435
- Papaya mosaic virus (PMV), 435
- Papaya ring spot (PRV), 435, 573
 control strategies of, 435
- Papaya ring spot virus (PRSV), 435, 444
- Parameters, physicochemical, 549
- Paraquat, 136
- Parasites, nematode, 179–181
- Parasitization of syrphid larvae, 248
- Parasitoid(s), 370, 477
 artificial diets for, 370
 in biological control, unisexual, 683–684
 of caterpillars, 133
 in farming systems, 387–390
 hymenoptera, 248
 pathogens of hymenopterous, 453
- Parasitosis, psychogenic (delusory parasitosis (DP), delusions of parasitosis, Ekblom's syndrome), 138–140
 causes of, 139
 delusions of, 138
 hallucinogens, effect of, 139
 role of entomologists in
 dealing with, 139
 symptoms of, 138–139
- Parathion, 69
- Paraxon
 enzyme on, action of, 69
 reaction of enzyme with, 70
- Paresthesia, 139
- Parlatoria oleae* (olive scale), 425
- Parthenolecanium persicae*
 (grapevine scale), 200
- PASS. *See* Pivot-attached sprayer systems.
- Passalora personata*, 469
- Passer domesticus* (house sparrows), 52, 347
- Pasteur effect, 187
- Pathogen(s)
 adventive, 405
 detection of
 biodiagnosis, 492
 culture indexing, 490, 492
 serodiagnosis, 492
 virus indexing, 492
 elimination of, 492, 675, 678
 fungal, 432
 greenhouse plant
botrytis cinerea, 214–215
 bacteria, 215–216
 managing, 214–217
 powdery mildew, 215
 viruses, 216–217
 water molds (*pythium* and *phytophthora*), 216
- of hymenopterous, 453
- plant, 405, 675–678
 diseases and, 676–677
 eliminations of, 573, 675–678
 of fruit crops, 676–677
 organs infected, 677–677
 postharvest, 656
 root-borne, 655
 soil-borne, 656
 soil-borne fungal, 644
 soybean fungal, 624
 survival of, 557–559
streptomyces soil rot, 656
 systemic, 655
- Pathogenicity, virus, 691
- PBNV. *See* Peanut bud necrosis virus.
- PCR. *See* Polymerase chain reaction.
- PDB. *See* Phytoplasma dieback.
- PDV. *See* Prune dwarf virus.

- Pea (*Pisum sativum*) diseases
 bacterial
 blight, 459
 pink seed, 459
 ecology and control of, 457–459
 fungal, 459
 downy mildew (*Peronospora viciae*),
 56, 457
 fusarium wilt (*Fusarium oxysporum*), 457
 gray mold (*Botrytis cinerea*), 459
 mycosphaerella blight
 (*Mycosphaerella pinodes*), 457, 458
 powdery mildew (*Erysiphe pisi*), 75, 76,
 437, 457–458
 root rot, aphanomyces and
 fusarium, 458
 seedling blight, 458
 white mold (*Sclerotinia sclerotiorum*), 459
 management, strategies for, 459
 nematodes, 459
 viruses, 459
- Pea (*Pisum sativum*) insects
 ecology and control of, 461–463
 foliage feeders, 461
 leafminers, 461–462
 leaf weevil, 461
 phloem extractors, 462–463
 aphid, 462
 seed feeders, 463
 weevil, 463
- Pea leafminer (*Liriomyza huidobrensis*), 461
- Pea, wild (*Pisum fulvum*), 463
- Peach canker (perennial canker), 467
- Peach (*Prunus persica*) diseases
 brown rot, 465–466
 causal organisms, 466
 ecology and control of, 465–468
 leaf curl, 465
 characteristics of, 466
 perennial canker, 467–468
 characteristic of, 467
 scab, 468
- Peach fruit moth (*Carposina sasakii*), 474
- Pea leafminer (*Chromatomyia horticola*), 462
- Pea leaf weevil (*Sitona lineatus*), 461
- Peanut (*Arachis hypogaea*) diseases
Cylindrocladium black rot, 470
 ecology and control of, 469–473
 leaf spots, early and late, 469
 rosette, 470
 rust, 469–470
 stem rot, 470
- Peanut bud necrosis virus (PBNV), 472
- Pea pod borer (*Etiella zinckenella*), 463
- Pear codling moth (*Cydia pyrivora*), 474
- Pear insects
 control, biological
 parasitoids, 477
 predators, 477
 ecology of, 474–477
 pest control
 insecticidal, 474–476
 mating disruption control, 476
- Pear psylla, 474, 475
- Pea weevil (*Bruchus pisorum*), 463
- Pecan (*Carya illinoensis*), 478
- Pecan aphid (*Monellia caryella*), 478
- Pecan insects, 478–480
 control of, 479
 ecology of, 478–479
- Pecan weevil (*Curculio caryae*), 478
 role of, 479
- Pectinophora gossypiella* (pink bollworm),
 554
- Pectinophora gossypiella*, 336
- Pentachlorophenol (PCP), 251, 253, 719
 immunotoxic effects in man, 253
 immunotoxicity of, 251
- Percent colonized vascular bundles
 (%CVB), 644
- Perennial canker (valsa canker, cytospora
 canker, and peach canker), 467
- Peridomestic pests, 244–245
 habitats, 246
 infesting, 245
 invading, 246
 management strategies for, 244
- PER. See Pesticide exposure record.
- Personal protective equipment (PPE), 725
- Permethrin (synthetic repellents), 353
- Peronospora parasitica*, 56
- Peronospora viciae* (downy mildew), 56, 457
- Persistent organic pollutants (POPs), 147,
 230, 363, 412, 481–483
 pesticides, 483
 protocol of, 365
- Pest(s)
 animal, monitoring of, 61
 in spring barley, 61
 in winter wheat, 61–62
 arthropod, 389–390, 474
 of cherry, 79
 control, use of IPM, 686
 of grape, 207
 management of, 686
 of strawberries, 630–632
 biocontrol of feeding
 external, 42–43
 internal, 42–43
 biological control of stored-product, 42–44
 containment program, 653–654
 control
 bacterial, 30–32
 concept, 516
 genetic engineering in, benefits of, 45
 hazardous alternatives, 310–312
 hazardous techniques for, 311
 policy, 230
 practices, 158–159
 repressive, 281
 systematics and biological, 661–663
 control method, 516–518
 and control tactics, insect and mite,
 544–546
 damage, effect of weather events on, 88
 domestic
 habitats, 246
 infestation of, 245
 overwintering, 245
 prevention and elimination
 strategies for, 245–246
 social, 245
 solitary, 245
 effects of different mulches on, 361–362
- [Pest(s)]
 eradication, 484
 animal movement control, 485
 fly trapping, 486
 screwworm as model, 484
 sterile insect technique, 484–485
 surveillance and prophylaxis, 485–486
 foliar, 131–132
 identification of, 261
 infestation of domestic, 245
 infestations of, 517
 influence on, 360
 insect ecology, 131–132
 management
 approaches to, 583
 implementing integrated, 259–260
 importance of integrated, 273
 natural enemies monitoring for, 377–381
 protected crops, 544–547
 satellite imagery in, 594–596
 strategies, 244–245, 615
 of tea, 673–674
 temperate-climate fruit crop, 675–678
 migration and movement, effects of, 259
 and natural enemies in stored products,
 examples, 43
 occurrence of, 60, 62
 outbreak, examples, 598–600
 outbreaks and climate, 87
 current trends, 88
 future projections, 89
 response to climate variables, 87
 and pesticide management
 in development cooperation, 144–148
 international training program, 147
 IOMC involvement in capacity
 building for, 146
 multilateral environmental
 agreements, 148
 reduction policies in developing
 countries, 144
 sustainable agriculture and IPM, 147
 and pesticide safety in homes and
 gardens, 102
 phytosanitary quarantine as, 516–518
 peridomestic, 244–245
 habitats, 246
 infesting, 245
 invading, 246
 management strategies for, 244
 prevention and elimination strategies for
 domestic, 245–246
 resurgence, 597–600
 causes of, 597
 factors influencing, 598
 management of, 600
 soil-dwelling, 121
 species, non-indigenous, 404–406
 of tea, 672–673
 tolerance in crops, 487–488
 turfgrass, 268
 upsets, 597–598
- Pest-free planting stock
 cost-benefit analysis of, 493–494
 development of, 493
 elimination of pathogens from, 492
 heat treatment (thermotherapy), 492

- [Pest-free planting stock]
 maintenance and prevention of
 reinfestation of, 492
 meristem tip culture, 492
 micrografting, 492
 mother plants, selection of, 490
 pathogens, detection of
 biodiagnosis, 492
 culture indexing, 490, 492
 serodiagnosis, 492
 virus indexing, 492
 regulatory control, 493
 selection of, 490–494
- Pesticide(s), 626
 agricultural
 in developing countries, 626–629
 strategies for reducing
 risks with, 626–629
 applications, legal aspects of, 307–309
 banning exports of banned, 571
 biodegradation, 161–162
 chemistry of, 67, 68–69
 choosing strategy, 628
 classification of, 542
 cross-resistance to
 cost of, 128–129
 future prospects, 129
 mechanisms of, 128
 definition of, 67
 destruction of natural enemies
 by, 385–386
 detection of, 218, 221
 ecological effects of, 538–539
 effects on wildlife, 539
 and energy use, 157
 enhanced microbial degradation
 of, 161–162
 environmental behavior of, 576
 evaluation of, 67
 exposure
 epidemiological studies, 724
 health risks of, 722–723
 toxicological manifestations of, 723–724
 worker protection from, 723
 in workers, symptoms of, 724
 formulation of, 548, 577
 quality control, 548–550
 found in monitoring programs in Europe
 and USA, 218
 in groundwater, 218–221
 hazard communication symbols of, 224
 hazard reduction, strategies
 critique of industry claims, 499
 global safe use campaign, 499
 making safe use work, 500–501
 health impacts in developing countries,
 228–231
 inhalation of microbial, 34
 limitations of safe use strategies, 581–582
 management education program, 496
 manufacture, 153, 159
 energy invested in, 157–158
 energy use in, 153–155
 metabolic activation of, 723
 microbial degradation of, 162
 microbial growth and adaptation, 161
 mode of using, improving, 627
- [Pesticide(s)]
 neurotoxic, 396
 obsolete
 guidelines related to, 415
 management of, 412
 reasons for accumulation of, 413
 stocks, 413
 types of, 414
 persistence of, 551
 as persistent organic pollutants, 73–74
 and pests safety in homes and
 gardens, 103
 physicochemical properties of, 576–577
 pictograms, 223
 poison, causes of, 590
 poisoning, acute, 228–229
 poisoning surveillance,
 national, 367–369
 as pollutants, 538
 pollution
 direct and indirect effects of, 538
 ecological effects of, 538–539
 effects on wild life, 539
 future potential, 539–540
 potential impacts of, 543
 prescriptive use of, 542–543
 production
 energy components of, 154
 energy cost/use in, 153–155
 production and use, energy in, 157–159
 qualifications of prescribers, 542
 rate of movement of, 577
 reduction strategy, 626–627
 risk function, 626
 concept of, 626
 role in, 155, 548
 safe use
 country's point of view, 581–583
 farmer's association point of
 view, 585–587
 regulator's point of view, 591–593
 suitability of, 67
 toxicodynamics of, 723
 use of, 67
 reducing, successes in, 551–552, 592
 virus
 anticrosia gemmatilis (AgMNPV),
 689, 690
 chirstoneura fumiferana
 (CfMNPV), 689
 cydia pomonella (CpGV), 689
 helicoverpa zea (HzSNPV), 689
 lymantria dispar (LdMNPV), 689
 neodiprion sertifer (NeseNPV), 689
 orgyia pseudotsugata (OpMNPV), 689
 spodoptera exigua (SeMNPV), 689
 trichoplusia ni (TnMNPV), 689
 for vegetables, consumption and
 cost of, 687
 workers, 722
- Pesticide exposure record
 (PER), 283, 285, 287
- Pesticides labels, 495–497
 attributes of
 environment, 224–225
 message, 223–224
 reader, 224
- [Pesticides labels]
 components
 colors, 223
 signal words, 223
 symbols, 223
 development of, 495–497
 initiatives, 497
 global harmonization of, 225–226
 types of
 primary, 497
 special local need (SLN) label, 497
 supplemental distributor, 497
- Pest management alternatives, 263–266
 biological control, 264–265
 beneficial insects and mites, 265
 disease-causing microorganisms, 265
 cultural methods
 insect-resistant and endophyte-enhanced
 grasses, 265
 turfgrass selection, 263
 insecticides/acaricides, 266
- Pest, vertebrate
 physical barriers for
 bats, 513
 birds, 511
 opossums, 513–514
 rabbits, 514
 raccoons, 513–514
 rodents, 511–513
 skunks, 513–514
 white-tailed deer, 514–515
- Petroleum oil, 416
 role of, 420
- Phaenicia sericata*, 245
Phaeoisariopsis personata, 469
Phasianus colchicus, 347
Pheidole megacephala, 109
Pheidologeton affinis, 109
 Phenoxy herbicides, 253
 immunotoxic effects in man, 253
- Phenyl organophosphates, 68
 Phenylpyrazole, 398
- Pheromone(s), 507
 applications in
 attracticide, 508–509
 mass trapping, 333, 507–508
 mating disruption, 509
 monitoring, 507
 applications of insect, 502
 lure, 502, 503
 synthetic, 34
 trapping system
 basic components of, 502
 merit and shortcoming of, 505
 types of, 503, 504
 use in IPM programs, 503
- PHIs. *See* Preharvest intervals, 496
- Phloem extractors, 462
- Phloeotribus scarabaeoides*
 (olive bark beetle), 425
- Phoma lingam*, 57
Phoma medicaginis, 7
Phoracantha semipunctata, 299
Phormia regina, 245
- Phosphorylation, 69
- Phosphorothiolate fungicides, 345
- Photostable pyrethroids, uses of, 72

- Photosynthesis inhibitors, 697
 Photosynthetically active radiation (PAR), 17, 608
 Photosystem II (PSII)
 herbicides, 170
 inhibitors, 170
Phyllosticta maydis, 408
Phyllotreta cruciferae, 58
Phyllotreta striolata, 504
Phyllotreta vittula (cereal flea beetle), 61
 Phytochemicals, 723
 Phytohemagglutinin (PHA), 253
Phytonemus pallidus (cyclamen mite), 631, 634
Phytophthora spp., 216
Phytophthora infestans, 595
Phytophthora infestans, 47
 Phytoplasma dieback (PDB), 2
 Phytoplasma diseases, 436
 Phytoplasma mosaic (PM), 436
 symptoms of, 436
 Phytoplasma yellow crinkle (PYC), 436
 Phytosanitary disease control system, 517
 Phytosanitary quarantines
 basis for, 516
 benefits and costs of, 516
 effectiveness of, 517–518
 intention of, 516
 as pest control method, 516–518
 programs
 challenges facing, 516–517
 effectiveness of, 518
Phytoseiulus persimilis, 453, 544, 634
 Phytotoxicity, surfactant-induced, 2
 Phytozoophagy, 172
Pieris brassicae (white butterfly), 132
 Pigment production inhibitors, 697
 Pigweed (*amaranthus retroflexus*), 125
 Pineapple sett rot
 (*ceratocystis paradoxa*), 644
 Pine voles, 694
 Pink bollworm (*pectinophora gossypiella*), 554
Pinus palustris, 110
 Piperonyl butoxide, 345
Pisum fulvum (wild pea), 463
Pisum sativum (pea) diseases
 bacterial
 blight, 459
 pink seed, 459
 ecology and control of, 457–459
 fungal, 459
 downy mildew (*peronospora viciae*), 56, 457
 fusarium wilt (*fusarium oxysporum*), 457
 gray mold (*botrytis cinerea*), 459
 mycosphaerella blight
 (*mycosphaerella pinodes*), 457, 458
 powdery mildew (*erysiphe pisi*), 75, 76, 737, 457–458
 root rot, aphanomyces and
 fusarium, 458
 seedling blight, 458
 white mold (*sclerotinia sclerotiorum*), 459
 management, strategies for, 459
 nematodes, 459
 viruses, 459
Pisum sativum (pea) insects
 ecology and control of, 461–463
 foliage feeders, 461
 leaf weevil, 461
 leafminers, 461–462
 phloem extractors, 462–463
 aphid, 462
 seed feeders, 463
 weevil, 463
 Pivot-attached sprayer systems (PASS), 64
Plagiohammus spp., 97
Planococcus citri (citrus mealybug), 504
 Plant(s)
 breeding, 519–523
 genetics of resistance and, 197–199
 strategies, 522
 diseases
 economic importance of, 574
 elimination of, 574
 domestication, centers of, 151
 feeding
 benefits, 172
 by predators, 172
 risks of, 172
 significance of, 172
 pathogens
 diseases and, 676–677
 eliminations of, 573, 675–678
 of fruit crops, 676–677
 organs infected, 677–677
 Plant health care (PHC), 298
 Plant–insect interactions and ecological impact, 107–108
 Plant-parasitic nematodes, 392
 Plant phenological indicators (PPIs), 299
Plasmiodiophora brassicae, 56, 438
Plasmodium falciparum, 540
Plasmodium halstedii, 647
 Plastic mulch, reflective, 606
 effectiveness of, 607
 efficacy of, 606
Platynota stultana, 212
Plodia interpunctella (Indian meal moth), 245, 507, 553
Plutella xylostella (diamond-back moth), 132, 242, 335, 387, 553, 554
 PmeV. *See* Papaya meleira virus.
 PM. *See* Phytoplasma mosaic.
 PMV. *See* Papaya mosaic virus.
 PNRSV. *See* Prunus necrotic ring spot virus.
Poa pratensis (kentucky bluegrass), 303, 304, 306
Pochonia chlamydosporia, 662
Podisus maculiventris, 371
Podosphaera oxycanthae, 75
 Poison baits
 for arthropods
 advantages and limitations, 527
 composition, 527
 efficacy, 527, 528
 future needs, 528–529
 for rodents
 advantages and limitations, 528
 composition and distribution, 527–528
 efficacy, 529
 selectivity and attraction of, 528
 Poisoning severity score (PSS), 283
 Poisonous arthropods, 531–537
 apidae. bees, 536
 arachnida, 531–534
 scorpions, 531
 spiders, 532–533
 ticks, 533–534
 chilopoda, centipedes, 534
 diplopoda, millipedes, 534
 formicidae. ants, 535–536
 hemiptera. true bugs, 536
 hymenoptera, 535
 insecta, 535–537
 lepidoptera. caterpillars and moths, 534–535
 Pookah Boeng, 645
Pollenia rudis (cluster fly), 245
 Pollinators, density of, 155
 Pollutants, 538
 Pollution of environment,
 pesticides, 538–540
 direct and indirect effect of, 538
 ecological effect of, 538–539
 effects on wild life, 539
 future potential, 539–540
 Polychlorocycloalkanes, 397, 398
 Polyethylene terephthalate (PET), 504
 Polygenic resistance, 520, 522
Polygonum aviculare (prostrate knotweed), 303
 Polymerase chain reaction (PCR), 346
 Polymorphic marker loci, 16
 Polymorphism, DNA, detection of, 15
 Polynesian rat (*rattus exulans*), 562
Polyphagotarsonemus latus (broad mite), 444
 Polyvinyl chloride (PVC), 191
Pongamia pinata (pongram tree), 416
Popillia japonica (Japanese beetle), 30
 POPs. *See* Persistent organic pollutants.
Populus dakhuanensis, 21
Poria hypobrunnea, 670
Postelectrotermes militaris, 673
Potamogeton crispus, 641
Potamogeton ochreateus, 641
 Potato
 virus-resistant, in Mexico, 47–50
 Potato leafhopper (*empoasca fabae*), 11
 Potato spindle tuber viroid (PSTVd), 449
 Potato virus (PV), 449, 450
 Potyvirus, 435
 Powdery mildew (*erysiphe pisi*), 75, 76, 437, 457–458
 PPE. *See* Personal protective equipment.
 Prairie voles, 694
Prays oleae (olive moth), 425, 426
 Predators, 370, 477
 artificial diets for, 370
 Preharvest intervals (PHIs), 496
 Principal component analysis (PCA), 249
 Prior informed consent (PIC),
 history of, 570–571
Prokelisa marginata, 256
Prostephanus truncates, 42
 Prostrate knotweed (*polygonum aviculare*), 303
 Prostrate spurge (*chamaesyce humistrata*), 303

- Protein endotoxin, 31
 Protein toxins, crystal, 31
 Protoporphyrinogen oxidase (PPO), 135
 Protozoan parasites, 453–454
 PRSV. *See* Papaya ring spot virus.
 Prune dwarf virus (PDV), 77
Prunus avium (sweet cherry), 75, 79.
 See also Cherry.
Prunus cerasus (sour cherry), 75, 79
 See also Cherry.
 symptoms on, 76
Prunus domestica, 387
 Prunus necrotic ring spot virus
 (PNRSV), 77
Prunus persica (peach) diseases
 brown rot, 465–466
 causal organisms, 466
 ecology and control of, 465–468
 leaf curl, 465
 characteristics of, 466
 perennial canker, 467–468
 characteristic of, 467
 scab, 468
Prunus spinosa, 387
 PRV. *See* Papaya ring spot.
 PSII. *See* Photosystem II.
Pseudalacaspis duplex, 672
Pseudaleptomastix squammullata, 211
Pseudaletia unipuncta, 390
Pseudaphycus angelicus, 211
Pseudococcus longispinus (long-tailed
 mealybug), 200
Pseudococcus maritimus, 207
Pseudococcus viburni, 65
Pseudomonas fluorescens, 32
Pseudomonas solanacearum, 573
Pseudomonas syringae, 76, 459, 649
Pseudothraupis vastans, 109
Pseudothraupis wayi, 109
Pseudozyma flocculosa, 215
 Psychogenic parasitosis (delusory parasitosis
 (DP), delusions of parasitosis,
 Ekbohm's syndrome), 138–140
 causes of, 139
 delusions of, 138
 hallucinogens, effect of, 139
 role of entomologists in dealing with, 139
 symptoms of, 138–139
Pteromalus puparum, 133
Puccinia arachidis, 469
Puccinia kuehnii, 645
Puccinia melanocephala, 645
Pulvinaria psidii, 672
 Purple nutsedge (*Cyperus rotundus*), 187
 PYC. *See* Phytoplasma yellow crinkle.
Pycnonotus jocosus (bulbuls), 347
 Pyrethrins, 71
 Pyraflufen-ethyl, 135
 Pyrethrin, 398
 Pyrethroid(s), 107, 128, 129, 396, 398
 generations of, 71
 insecticides, 67
 photostable, uses of, 72
 synthetic, 71
 Pyriproxyfen, 85
Pythium aphanidermatum, 216
Pythium ultimum, 362, 408
 Quackgrass (*Elymus repens*), 304
 Quantitative trait loci (QTL), 14, 520
 Quarantines
 phytosanitary
 basis for, 516
 benefits and costs of, 516
 effectiveness of, 517–518
 intention of, 516
 as pest control method, 516–518
 programs, 516–518
 plant, problems in implementing, 517
Quelea quelea (red-billed quelea), 52
 Queletox[®], 52

Radinaphelenchus cocophilus, 573
 Ragweed (*Ambrosia artemisiifolia*), 58, 125
Ralstonia solanacearum, 215
Rana nigromaculata, 40
Rana rugosa, 40
 Random amplified polymorphic DNA
 (RAPDs), 199
 Ratoon stunting disease (RSD), 644
Rattus exulans (polynesian rat), 562
Rattus flavipectus, 562
Rattus norvegicus (Norway rat), 562
Rattus rattus (roof rat), 562
 Receptors
 acetylcholine
 nicotinic, 398–399
 postsynaptic, 399
 muscarinic, 398
 Red-billed quelea (*quelea quelea*), 52
 Red deer (*Cervus elaphus*), 347
 Red imported fire ant in crop insect
 control, 113–114
 Red locust (*Nomadacris septemfasciata*), 319
 Red mite (*Panonychus ulmi*), 598
Reduvius personatus, 536
 Red-winged blackbirds
 (*Agelaius phoeniceus*), 52
 Refugia
 aspects of, 272
 importance of structured, 272
 strategy, high-dose, 554
 Reglone[®], 641
 REI. *See* Restricted-entry interval.
 ReJeX-iT[®], 54
 Remote sensing, 652
 of electromagnetic spectrum, 594
 technology uses in pest management,
 594–595
 of vegetation, 594, 595
 Reniform nematodes, 436, 656
 REN. *See* Restriction enzyme.
 Repelin[®], 422
 Repellents, 694
 chemical, 353
 primary, 54
 secondary, 54
 Repellents, synthetic, 353
 N,N, diethyl-m-toluamide (DEET), 358,
 717
 concentrations of, 353
 formulations of, 353
 permethrin, 353
 Replicase (RP), 435

 Resistance
 to Bt transgenic plants, 553–554
 durability of, 520–522
 fungicide, feature of, 345
 genes, pyramiding of, 522
 genetics and mechanisms of, 553–554
 geographical deployment of different
 genes for, 522
 management strategies, 554
 mechanisms, 198
 metabolic, features of, 344
 monogenic, 520, 522
 plants, 133
 polygenic, 520, 522
 rapid diagnosis of, 346
 types of, 520
 Restricted-entry interval (REI), 723
 Restriction enzyme (REN), 690
 Restricted use pesticides (RUPs), 307,
 393, 583
 Restriction fragment length polymorphisms
 (RFLPs), 199
 Resurgence
 secondary pests, 597–600
 of insect and mite pests, 599
 examples of, 598
 outbreaks, 598
 management of, 600
 pesticide-induced, 598
 Return-polyacrylamide gel electrophoresis
 (R-PAGE), 451
 Reverse transcription-polymerase chain
 reaction (RT-PCR), 451
Rhabdophis tigrinus, 40
Rhagoletis cerasi, 84
Rhagoletis cingulata, 80
Rhagoletis fausta, 80, 84
Rhagoletis indifferens, 80, 84
Rhizoctonia solani (soil-borne fungus), 9,
 57, 408, 459
Rhizomonas suberifaciens, 317
Rhizopus oryzae, 656
Rhizopus stolonifer, 437, 656
Rhopalosiphum maidis, 644
Rhynchites cribripennis
 (twig cutter beetle), 425
Rhyzopertha dominica, 43
 Rice (*Oryza sativa*) diseases, 556
 control options
 chemical, 560–561
 cultural practices, 560
 resistant varieties, 560
 development of, 560
 ecology and control of, 556–561
 impact of, 556
 survival of pathogen, 557–559
 symptoms of, 557–559
Rickettsiella phytoseiuli, 455
 Rid-A-Bird[®], 52
Riptortus clavatus, 463
Riptortus dentipes, 106
 Rock doves (*Columba livia*), 347
 Rock pigeons (*Columba livia*), 52
 Rocky mountain locust
 (*Melanoplus spretus*), 320
 Rodent(s), 567
 control measures of, 562

- [Rodent(s)]
 exclusion, 562–565
 concept of, 565
 future prospects of, 564
 pests
 management of, 563–564
 physical abilities and characteristic features of, 564
 population, impact of, 562
 proofing, 563
 and slugs, 615–616
- Rodenticides, 567
 alternatives to, 569
 anticoagulant-type, 528
 biological, 569
 fast-acting or acute, 567–568
 fluoroacetamide (1081), 567
 sodium fluoroacetate (1080), 567
 strychnine, 567–568
 zinc phosphide, 568
 future prospects of, 569
 slow-acting (chronic), 568
 alpha-chloralose, 568
 anticoagulants, 568
 bromethalin, 568
 calciferol (vitamin D), 568
 types of, 694–695
- Rodent-proof materials, 565
- Rodolia cardinalis*, 377
- Roof rat (*rattus rattus*), 562
- Root-borne pathogens, 655
- Root crop insects
 control
 biological, 132–133
 cultural, 133
 insecticidal, 133–134
 ecology and control of, 131–134
- Root-knot nematode, 656
- Ro-Pel[®], 54
- Rotilenchus reniformis*, 436
- Rotterdam convention, 570–572
- Rotylenchulus reniformis*, 656
- Rouging
 applications of, 573–574
 costs and benefits of, 574
- R-PAGE. *See* Return-polyacrylamide gel electrophoresis.
- RSD. *See* Ratoon stunting disease.
- RT-PCR. *See* Reverse transcription-polymerase chain reaction.
- Runoff
 geographical and hydrological characteristics affecting, 578
 management practices, 579
 meteorological factors affecting, 578
 processes, testing methods, 578–579
 theory and factors affecting, 576–578
- Rural filariasis, 179
- Rye trips (*limothrips denticornis*), 61
- Rynchops niger*, 184
- Saccharum officinarum*, 643
- Saccharum spontaneum*
 (tiger grass), 187, 643
- Safety data sheets (SDS), 223
- Saissetia oleae* (black scale), 425
 cultures of, 569
- SAR. *See* Systemic acquired resistance.
- Satellite imagery in pest management, 652
 precision agriculture, 594
 remote sensing of electromagnetic spectrum, 594–595
 satellites and resolution, 595
 sensor generated information, 594
 use of satellite data, 595
 variable rate technology, 595
- SBLs. *See* Sterol biosynthesis-inhibiting fungicides.
- Scatterbird[®], 52
- Schistocerca gregaria*
 (desert locust), 320–321
- Schizaphis graminum*, 618
- Sciurus carolinensis* (grey squirrel), 347
- Sclerotinia* diseases, 73
- Sclerotinia sclerotiorum* (white mold), 57, 317, 459, 649
- Sclerotinia trifoliorum*, 7
- Sclerotium rolfsii*, 470
- Sclerotium wilt, 470
- Screwworm
 eradication of, 486
 as model for pest eradication, 484
 myiasis, 484
- Scurf pathogen (*monilochaetes infuscans*), 655
- Scutellista caerulea*, 427
- SDS. *See* Safety data sheets.
- Secondary pests, 597–600
 examples of, 598
 outbreaks, 598
 resurgence, 597–600
- Seed certification programs, 450
- Semaphore cactus (*opuntia spinosissima*), 33
- Semiochemicals, 332
 pheromones and, 334
 examinations of mass-trapping, 334
- SeMNPV. *See* *Spodoptera exigua*.
- Septoria pisi*, 459
- Septoria tritici*, 522
- Serica assamensis*, 672
- Serodiagnosis, 492
- Serpentine leafminer (*liriomyza trifolii*), 462
- Serratia entomophila*, 31
 culture and control of, 31
- SERVO[®], 420
- Setaria faberi*, 126
- Setaria glauca* (foxtail), 188
- S-ethyl dipropylcarbamothioate (EPTC), 162
- SEVIN, 551
- Sex hormones, determination of, 238
- Sheep predation, 602–604
 impact of predators, 603
 predators and habits, 602–603
 cougars, jaguars, and bears, 602–603
 coyotes, 602
 domestic and feral dogs, 602
 wolves, 602
 protective measures
 fencing, 603
 hunting and trapping, 604
 management, 603
- [Sheep predation]
 mobile protectors (guard animals), 603, 604
- Shoot beetle (*tomicus piniperda*), 290, 654
- Shoot fly (*atherigona soccata*), 618
- Sigatoka in banana, black, 573, 574
- Silverleaf whitefly (*bemisia argentifolii*), 652
 host for, 606
 incidence of, 608
 infestations of, 607
 management of, 606–609
 response, to reflective plastic and wheat straw mulch, 606, 608
 cucumber, 606–607
 pumpkin, 606
 spectral reflectance, 608
 squash, 608
 symptoms of, 606
- Simple sequence repeats (SSRs), 199
- Siphonius phillyrae* (ash whitefly), 299
- SIT. *See* Sterile insect technique.
- Sitona lineatus*, 461
- Sitotroga cerealella*, 384
- Sitophilus granarius* (granary weevil), 44
- Slow-acting (chronic) rodenticides
 alpha-chloralose, 568
 anticoagulants, 568
 bromethalin, 568
 calciferol (vitamin D), 568
- Slugs and rodents, 615–616
- Snail, African (*achatina fulica*), 33
- Sodium dodecyl sulfate-polyacrylamide gel electrophoresis (SDS-PAGE), 690
- Sodium fluoroacetate (1080) (acute rodenticides), 567
- Soil
 carbon levels of, 616
 characteristics of, 577
 environment, complexity of, 429
 erosion, 122, 616
 fumigation, 393
 infiltration by pesticides, 610–613
 insect populations, 122
 management practices
 beneficial, 616
 effects of, 615
 insects, 615
 slugs and rodents, 616
 moisture, 407
 and non-soil factors,
 classification of, 611
 pests, 132
 populations, assessment of, 122
 quality effects, 125
 survival of rice in submerged, 188
 tolerance mechanism of weeds to flooded, 187
 water conservation, 616
- Soil-active insects, 262
- Soil amendments, organic, 428–430
 effect of, 430
 efficacy of, 429
 impacts of, 428
 mechanisms of action, 429
 and plant resistance, 428
 use of, 429

- Soil-applied systemic insecticides, 665, 666, 667
- Soil-borne fungal pathogens, 644
- Soil-borne fungi, 681
- Soil-borne fungus (*rhizoctonia solani*), 9, 57, 408, 459
- Soil-borne pathogens, 656
- Soil-dwelling pests, 121
- Solanum tuberosum*, 595
- Solenopsis geminata*, 109
- Solenopsis invicta*, 110, 113, 183, 404
- Solenopsis richteri*, 183
- Solid wood packing materials (SWPMs), 289–290
use of, 25, 289
- Somaclonal variation, 492
- Somatic cell hybridization, 492
- Sorghum bicolor*
insects
control, biological and chemical, 620, 621
ecology and control, 618–621
pest management, 618–621
- Sorghum halepense* (Johnson grass), 187
- Sorghum mosaic virus, 643
- South American moth (*cactoblastis cactorum*), 34
- Soybean aphid (*aphis glycicinis*), 654
- Soybean (*glycine max*)
diseases, ecology and control of, 623–624
fungal pathogens of, 624
- Soybean stem miner (*melanagromyza sojae*), 462
- SPaV. *See* Strawberry pallidosis associated virus.
- SPCSV. *See* Sweet potato chlorotic stunt virus.
- Special local need (SLN) label, 497
- SPFMV. *See* Sweet potato feathery mottle virus.
- Spider mite, two-spotted (*triticum urticae*), 631–632
- Spinosad[®], 426
- Spinosoid, 398
- Spinosyn, 398
- Spiny mouse (*acomys coharinus*), 562
- Spodoptera exigua* (SeMNVPV), 635, 689
- Spodoptera frugiperda*, 63, 113
- Spodoptera litura* (cluster caterpillar), 333, 504, 631
- Sprinkler irrigation, 64
- Squash, zucchini, 608
- Starlicide[®], 52
- Starlings (*sturnus vulgaris*), 52
- Stegobium paniceum*, 509
- Steinerema feltiae*, 49
- Steinernema carpocapsae*, 453
- Steinernematid nematodes, 453
- Stemphylium lycopersici*, 438
- Stenodiplosis sorghicola*, 618
- Sterile insect technique (SIT), 484–485
aspects of, 486
- Sterna nilotica*, 184
- Sterol biosynthesis-inhibiting fungicides (SBIs), 73
- Sterol inhibitors (SIs), 77
- Sticky or meleira diseases, 435
- Sticky trap, 502
- Stored-product pests, biological control of, 42–44
externally feeding, 43
factors, 42
internal feeding, 42–43
- Strawberry aphid (*chaetosiphon fragaefolii*), 630–631, 635
- Strawberry arthropods
ecology and control of, 630–631
insects and mites in California, ecology and control, 634–636
- Strawberry blossom weevil (*anthonomus rubi*), 630
- Strawberry bud weevil (*anthonomus signatus*), 630
- Strawberry pallidosis associated virus (SPaV), 635
- Strawberry root weevil (*otiorhynchus ovatus*), 631
- Straw fly (*chlorops pumilionis*), 61
- Streptomyces anulatus*, 429
- Streptomyces avermitilis*, 394
- Streptomyces ipomoeae*, 656
- Streptomyces* soil rot pathogens, 656
- Streptozotocin, 237
- Strobilurins, 215
- Strychnine (acute rodenticides), 52, 567–568
- Sturnus vulgaris* (starlings), 52
- Styrchnos nuxvomica*, 567
- Subsidies
diversity of, 373–375
resource, 373–374
- Sugarcane diseases
bacterial, 644–645
ecology and control of, 643–645
fungal, 644–645
seedcane quality, 643
viral and phytoplasmal, 643–644
- Sugarcane mosaic virus (SCMV), 643
- Sugarcane wireworms (*melanous okinawensis*), 333
- Sulfenimides, 73
- Sunflower (*helianthus annuus*) disease
ecology and control of, 647–650
foliar, 647–649
head rots, 649
seedling, 647
stalk and root, 649
- Sunspray[®] 7E, 420
- Sunspray 6E Plus[®], 420
- Sunspray Ultra Fine[®], 420
- Surface-feeding insects, 122
- Surfactant-induced phytotoxicity (plant cell membrane toxicity), 2
- Surfactants
adjuvants, 1
emulgen 913, 2
metabolism of, 2
non-ionic, 2
organosilicone, 1
toxicity, mechanism of, 2
- Surveillance
programs of USDA-APHIS-PPQ, 653
operations, 653–654
emergency program, Asian longhorned beetle, 653
- [Surveillance]
pest containment program, gypsy moth, 653–654
routine inspection, 654
techniques, 652
attractants, 652
assessment of pests or damage, direct, 652
remote sensing and digital imaging, 652
- Sustainability
agricultural, 37
of farming practices, 37–40
examples of potential bioindicators, 39
- Swedish International Development Cooperation Agency (SIDA), 146
- Sweet cherry (*prunus avium*), 75, 79.
See also Cherry.
- Sweet potato (*ipomoea batatas*), 655–656
- Sweet potato chlorotic stunt virus (SPCSV), 655
- Sweet potato diseases, ecology and control of, 655–656
- Sweet potato feathery mottle virus (SPFMV), 655
- Sweet potato weevils (*cylasformicarius elegantulu*), 110
- SWPM. *See* Solid wood packing material.
- Sympetrum frequens*, 40
- Synergism or potentiation
definitions of, 658
examples of, 659
- Synergy with microorganisms, 658–660
- Synthesis, lethal, 567
- Synthetic pyrethroids (SPs), 597
use of, 598
- Synthetic repellents
N,N, diethyl-m-toluamide (DEET), 358, 717
concentrations of, 353
formulations of, 353
permethrin, 353
- Syrphid larvae, parasitization of, 248
- Syrphidae* spp., 249
in Northern Italy, 249
as potential control of aphids, 247–248
role of, 249
- Systematics
and biological pest control, 661–663
challenges in, 663
methods in, 661
of organisms, 661
fungi, 662
insects and mites, 662–663
- Systemic acquired resistance (SAR), 435
- Systemic insecticides
benefits of, 665–666
characteristics of, 664–665
costs of, 666–667
- Tanglefoot[®], 54
- Taphrina deformans*, 465
- Taraxicum officinale*, 126, 303
- Tart cherry (*prunus cerasus*), 75, 79
See also Cherry.
symptoms on, 76

- Tea (*Camellia sinensis*)
 diseases
 ecology and control of, 668–671
 leaf, 669
 management of, 669–671
 of nursery plants, 669
 root, 668
 stem, 668–669
 insects, ecology and control of, 672–674
 pests of, 672–674
 Tebufenozide, 129
 Temperate-climate fruit crop pest
 management, 675–678
Teregra quadrangularis, 673
Teretrius nigrescens, 42
 Tergitol® (PA-14), 53
 Terminal insecticide concentration (TIC), 5
 Terrestrial invasive species
 characteristics of, 347
 economical and ecological impacts
 of, 348
 management of, 349
 mitigating impacts of, 347–349
 Tetrachlorvinphos, 68
 9,12-Tetradecadien-1-yl acetate (TDA), 508
Tetranychus cinnabarinus, 444
Tetranychus pacificus, 207
Tetranychus urticae, 634
Tetropium fuscum (brown spruce longhorn
 beetle), 290
 Thatch grass (*Imperata cylindrica*), 187
 Theileriosis, 14
Theocolax elegans, 43
 Thermal fog
 advantage of, 164
 applications of, 163–164
 Thermal foggers
 hand-carried, 164
 vehicle-mounted, 164
 Thidiazuron, 135
Thielaviopsis basicola, 459
 Thiophanates and benzimidazoles, 73
 Thiram, 694
Thlaspi arvense, 125
Thrips palmi, 472
 Thrombocytopenia, 141, 142
 TIC. *See* Terminal insecticide
 concentration.
 Tiger grass (*Saccharum spontaneum*), 187
 Tillage, 680–682
 conservation, benefits of, 407
 and cultivation, 680–682
 mechanical, 190–191
 advantages and disadvantages of, 190
 and other practices, 614
 prior to planting, 338
 ridge, 615
 strip, 614
 system
 conservation, 408, 615, 616, 680–681
 conventional, 407, 680
Tinocallis caryaefoliae (black aphid), 478
Tithonia diversifolia, 428
 TnMNPV. *See* *Trichoplusia ni*.
 Tobacco budworm (*Heliothis virescens*), 128
 Tobacco caterpillar
 (*Spodoptera litura*), 598
 Tobacco rattle virus (TRV),
 incidence of, 450
 Tobacco workers, 723
 Tomato fruitworm (*Helicoverpa zea*)
 (HzSNPV), 688, 689
 Tomato pinworm (*Keiferia
 lycopersicella*), 336
 Tomato spotted wilt virus (TSWV), 214,
 216, 472, 686
Tomicus piniperda (shoot beetle), 290, 654
 Total digestible nutrients (TDNs), 324
 Toxicants, 52
 acute, 528, 695
 chronic, 528
 lethal, 52, 53
 neuro, 395
 Toxic effect
 of fumigants, 194
 of herbicides, 47–49
 in man, immuno, 251–254
 carbamates, 252
 organochlorines insecticides, 251
 organophosphorous compounds, 251
 organotin compounds, 253
 pentachlorophenol (PCP), 253
 phenoxy herbicides, 253
 Toxicity
 immuno, 251
 of pentachlorophenol (PCP), 253
 of intrinsic, aldicarb, 665
 of herbicide resistant crops (HRCs)
 technology, 47
 Toxicodynamics of pesticide, 723
 Toxicosis, symptoms of, 528
 Toxins
 cry, 553
 crystal protein, 31
Toxoptera aurantii, 674
Toxotrypana curvicauda, 440
 Transgenesis (gene transfer), 15, 16
 Transgenic insecticidal cultivars, 553
 Transmissible spongiform encephalopathies
 (TSE), 14
 Trap(s)
 attract and kill, 426
 chromotropic, 247
 deployment, 504–505
 density, area size in treatment, 332–333
 designs, 502–504
 hand-net, 247
 lantern, 503
 leggett, 503
 malaise, 247
 mammal
 efficiency of, 326
 future needs, 327
 role in, 327
 strategies and concerns, 326–327
 types of, 326
 no-exit, 504
 non-sticky, 503
 placement considerations, 504
 sticky, 502
 types of, 332
 water, 503
 Trapping system, pheromones
 basic components of, 502
 [Trapping system, pheromones]
 merit and shortcoming of, 505
 types of, 503, 504
 use in IPM programs, 503
 Treesnakes, brown (*Boiga irregularis*), 347
Trialeurodes vaporariorum
 (whitefly), 114, 635
Triatoma magista, 536
Triatoma protracta, 536
Triatoma rubida, 536
 Triazine herbicides, 170
 Triazines, 72, 589
 Triazoles, 72
 Tribufos, 135, 137
 Tricarboxylic acid cycle (Krebs cycle), 567
Trichoderma aggressivum, 662
Trichoderma harzianum, 195, 472,
 662, 670
Trichoderma viride, 429, 472, 670
Trichogramma, 683
Trichogramma brassicae, 383, 388
Trichogramma carverae, 200
Trichoplusia ni (TnMNPV), 689
Trichosurus vulpecula (brush tail
 possums), 349
Triopis longicaudatus, 40
Trioza apicalis, 361
Tripsacum laxum (guatemala grass), 670
Triticum aestivum, 630
Triticum urticae (two-spotted spider mite),
 631–632
Trogoderma glabrum, 335, 508
Trybliographa rapae, 133
 Trypanosomiasis, 14
Tryporhyza nivella, 600
 TSWV. *See* Tomato spotted wilt virus.
 Tubo mata picudo (TMP), 333
 Turfgrass
 insect-resistant, 265
 integrated pest management (IPM)
 program for, 261–263
 management, effective, 263
 pests, 268
 weed management, 303
 mowing heights, 304
 Twig cutter beetle (*Rhynchites cribripennis*),
 425
Tylenchorhynchus cylindricus, 436
Tylenchulus semipenetrans, 428
Tylo alba (barn owl), 33
Tyrophagus putrescentiae, 453
 Ultra low volume (ULV) (cold fogging)
 applications, 164
 definition by environmental protection
 agency, 164
 spraying, 716
 aerial, 717
 technology, 164
 advantages of, 164
 Unisexuality, causes of, 684
 Unisexual parasitoids in biological
 control, 683–684
 United Nations Conference on
 Environment and Development
 (UNCED), 275, 363

- United Nations Convention on Biodiversity (UNCBD), 364
- United Nations Economic Commission for Europe (UN-ECE), 365
- United Nations Environment Programme (UNEP), 146, 147, 363
- United Nations Food and Agriculture Organization (FAO), 146
- United Nations Industrial Development Organization (UNIDO), 146, 147
- United Nations Institute for Training and Research (UNITAR), 146
- United Nations International Labor Organization (ILO), 365
- United States Department of Agriculture (USDA), 25, 382–383
- United States Department of Agriculture's Animal and Plant Health Inspection Service (USDA-APHIS), 25, 404
- United States Department of Agriculture Animal and Plant Health Inspection Service, Division of Plant Protection and Quarantine (USDA-APHIS-PPQ), 516, 653, 654
- surveillance programs of, 653
- Universal soil loss equation (USLE), 612
- Urban filariasis, 179
- Uromyces fabae*, 459
- USDA. *See* United States Department of Agriculture.
- USDA-APHIS. *See* United States Department of Agriculture Animal and Plant Health Inspection Service, Plant Protection and Quarantine.
- USDA-APHIS-PPQ. *See* United States Department of Agriculture Animal and Plant Health Inspection Service, Division of Plant Protection and Quarantine.
- USLE. *See* Universal soil loss equation.
- Ustilago scitaminea*, 645
- UV-reflective plastic mulches, 606
- mechanism of, 20
- Valsa canker (perennial canker), 467
- Variable rate technology (VRT), 595
- Vectors
- for filariasis, 180
 - insects, chemical control, 561
 - mosquito, 715
- Vegetable crop pest management, 686–688
- Vegetable leafminer (*liriomyza sativae*), 462
- Vegetation, natural
- ecological functions of, 387–389
 - management to improve parasitoids in farming systems, 387–390
 - on parasitoids, 387–389
- Vejuvis russelli*, 531
- Venturia canescens*, 43
- Vertebrate pests
- physical barriers for, 511–515
 - bats, 513
 - birds, 511
 - opossums, 513–514
 - rabbits, 514
 - raccoons, 513–514
 - [Vertebrate pests]
 - rodents, 511–513
 - skunks, 513–514
 - white-tailed deer, 514–515
 - Verticillium chlamydosporium*, 428, 429, 436, 662
 - Verticillium dahliae*, 649
 - Verticillium lecani*, 454
 - Vicia faba*, 49
 - Vigna unguiculata* (cowpea) insects
 - ecology and control of, 106–108
 - pests, 106–107
 - coleoptera, 106
 - heteroptera, 106–107
 - lepidoptera, 106
 - management of, 107
 - thysanoptera, 107
 - Vinegar flies (*drosophila* spp.), 636
 - Vineyards
 - Australian
 - beneficials and chemicals in, 200
 - management practices in, 200, 204–205
 - pest and control agents in, 201–203
 - biological control in California, 211
 - Virus(es)
 - associated to insects, 689
 - activity
 - biological, 691
 - insecticidal, 692
 - baculo, 689
 - activity in host insects, 692
 - commercial production of, in insect cells, 692
 - genetic modification of, 692
 - host specificity of, 691–692
 - pathogenesis, 689, 690
 - phylogeny of, 691
 - populations of, 691
 - use of, 689
 - for biological control of insects, 689–692
 - commercial production, 691–692
 - entomopathogenic, 658
 - insect
 - associated to, 689
 - identification of, 690–691
 - characterization of, 690–691
 - myxoma, 35
 - pathogenicity, 691
 - populations
 - analysis of, 691
 - increase diversity in, 692
 - pesticides
 - anticarsia gemmatalis* (AgMNPV), 689, 690
 - choristoneura fumiferana* (CfMNPV), 689
 - cydia pomonella* (CpGV), 689
 - helicoverpa zea* (HzSNPV), 689
 - lymantria dispar* (LdMNPV), 689
 - neodiprion sertifer* (NeseNPV), 689
 - orgyia pseudotsugata* (OpMNPV), 689
 - spodoptera exigua* (SeMNPV), 689
 - trichoplusia ni* (TnMNPV), 689
 - and viroid-free material, rapid propagation of, 450
 - and viroids
 - elimination, methods of, 449–450
 - [Virus(es)]
 - methods for the sensitive detection of, 451
 - Virus-free crops in field, management of, 450–451
 - Virus-free stock
 - management of, 449
 - potato crop as model, 449
 - steps for production of, 449
 - Virus-resistant potatoes in Mexico, assessment of transgenic, 47–50
 - Vitamin D (calciferol)
 - (chronic rodenticides), 568
 - mode of action of, 568
 - Vitis girdiana*, 207
 - Vitis labrusca*, 207
 - Vitis rupestris*, 207
 - Vitis vinifera*, 207
 - Volck Supreme Oil[®], 420
 - Voles
 - characteristics of, 694
 - and habitat, 693
 - management for orchards, 693–695
 - meadow, 694
 - monitoring for, 693–694
 - pine, 694
 - prairie, 694
 - Volks Supreme[®], 420
 - Voltage-gated sodium channels, 396, 398
 - VRT. *See* Variable rate technology.
 - Vulpes vulpes* (fox), 347
- Wasp, ichneumonid (*diapetimorpha introita*), 370
- Watermelon mosaic virus (WMV), 17
- Water trap, 503
- Weed(s)
- aerobic respiration in
 - hydrophytic, 187
 - mesophytic, 187
 - aquatic, chemical control, 640–641
 - in flowing water, 641
 - in New Zealand lakes, 641–642
 - in recreational lake, 641
 - aquatic, mechanical control
 - cutting and harvesting, 637
 - draglining/dredging, 638–639
 - rototilling, 638–639
 - biology and human health, 713
 - biomass, 124
 - and carbon dioxide, 712–714
 - characteristics of, 703
 - control
 - in agriculture, mechanical, 338–342
 - crop rotations for, 124–126
 - physiological adaptations and, 185–189
 - principal methods of,
 - biological, 275–276
 - strategies of, 709–710
 - through flooding, 187
 - control programs, biological, 696, 697
 - economic benefits of, 697
 - general procedures in, 697
 - implementation of, 696
 - development in fruit crops, 191
 - features of submerged, 187

- [Weed(s)]
 germination of, 186–187
 harvesting in small rivers and drains, 641
 infestation, economic impact of, 703
 invasive, 712–713
 management, 275, 303, 614–615
 biological control, 696
 biological control with other methods of, 276–277
 chemical control, 697–698
 cultural control, 703
 during fallow periods, 341–342
 in home landscape, 699
 implications for, 713–714
 integrated, 303
 mechanical control, 703
 no-till on, effects of, 408
 in nurseries, methods of, 705–707
 options for lawns and ornamentals, 699–701
 in ornamental nurseries, 705–707
 strategies in turf, 303
 turfgrass, 303
 management strategies, implications for, 708–711
 negative impacts of, 702
 perennial, 615
 pernicious, 187
 physiology of, 187
 population, 709
 ecology, 124
 science, 275
 seed dormancy, 708–711
 characteristics of, 709
 seasonal changes in, 709
 suppression, allelopathy role in, 703
 ventilation of water, 187–188
- Weed-crop interactions in agroecosystems, 712
- Weeding tools and implements, 339–340
- Weevil
 alfalfa, 114
 management of, 11
 banana, 110
 black vine, 631
 pea, 463
 pea leaf, 461
 pecan, 478
 role of, 479
 strawberry
 blossom, 630
 bud, 630
 root, 631
- Weevil, granary (*sitophilus granarius*), 44
- West Nile virus (WNV), 350, 351
 and mosquito control, 715–719
- Western flower thrips (*frankliniella occidentalis*), 214, 631, 636
 biological control of, 546
- Western tarnished plant bug (*lygus hesperus*), 106, 631, 635
- Wheat stem sawfly (*cephus cinctus*), 408
- Wheat straw mulch
 efficacy of, 606
 reduces aphid, 17
 in repelling aphids, 20
- Wheat trips (*haplothrips tritici*), 61
- White butterfly (*pieris brassicae*), 132
- White mold (*sclerotinia sclerotiorum*), 57, 317, 459, 649
- Whitefly
bemisia argentifolli, 595
bemisia tabaci gennadius, 598
trialeurodes vaporariorum, 635
- Whitefly, silverleaf (*bemisia argentifolii*), 652
 host for, 606
 incidence of, 608
 infestations of, 607
 management of, 606–609
 response, to reflective plastic and wheat straw mulch, 606, 608
 cucumber, 606–607
 pumkin, 606
 spectral reflectance, 608
 squash, 608
 symptoms of, 606
- WHO. *See* World Health Organization.
- Wild pea (*pisum fulvum*), 463
- WNV. *See* West Nile virus.
- Wolbachia*, 455
- Women, hormonal disruption in, 238
- Wood
 preservation, 719–721
 classical, 719
 future considerations of, 720–721
 history of, 719
- Wood preservatives
 classical, 719
 system
 management and control of, 720–721
 new generation, 719
- Worker pesticide exposure
 aspects of, 722–724
 complexities of, 724
 effects of, 724
 epidemiological studies and risk assessment, 724
 harmful manifestations of, 723–724
- [Worker pesticide exposure]
 health risks of, 722–723
 protection from, 723
 symptoms of, 724
 toxicological manifestations of, 723–724
- Worker protection standard (WPS)
 duties for employers, 725–726
 implementation of, 725
 Iowa survey, 727
 Michigan survey, 727
 requirements and definitions of, 725–727
- World Health Organization (WHO), 146, 181, 551, 558
- Worm
 black cut, 109, 408, 461
 corn ear, 408
 dusky wire, 630
 pin, 336
 pink boll, 554
 screw
 eradication of, 486
 as model for pest eradication, 484
 myiasis, 484
 sugarcane wire, 333
 tobacco bud, 114, 128
 tomato fruit, 688
 tomato pin, 336
- WPS. *See* Worker protection standard.
- Wuchereria bancrofti*, 179
- Xanthomonas albilineans*, 644
Xanthomonas campestris, 56, 215
- Xenoantiestrogens, 237
- Xenoestrogens, 237
- Xenohormones, 237
- Xestia c-nigrum*, 461
- Xylocoris flavipes*, 43
- Xylotrechus quadripes*, 97
- Yellow leaf syndrome (YLS), 644
- Yellow-type diseases in papaya, 436
- Zarhopalus corvinus*, 211
- Zeuzera pyrina* (leopard moth), 425
- Zinc phosphide (acute toxicants), 528, 567, 695
- Zinc salt (ziram), 54
- Zoonotic disease, 181
- Zoophytophagy, 172
- Zucchini squash, 608
- Zucchini yellow mosaic virus (ZYMV), 17

Encyclopedia of PEST MANAGEMENT VOLUME II



Praise for the **Encyclopedia of Pest Management**

"There are few people better qualified to put together a group of international experts on IPM than Dave Pimentel, and he has done an excellent job. This compilation of encyclopedic entries on various aspects of IPM should serve as a useful reference for the professional and student alike. The entries are both concise and informative."

— Dr. Paul C. Johnson, Department of Natural Resources, University of New Hampshire, Durham, U.S.A.

"...outstanding...timely...needed."

— Maurizio G. Paoletti, Department of Biology, Padova University, Italy

"This volume truly represents a tremendous effort...for libraries it can be recommended."

— Journal of Phytopathology

"...valuable...comprehensive...up-to-date references...easy to read...describes sophisticated, scientifically-based issues and management techniques for pest control."

— Mary Bomford, M.Sc., Ph.D., Principal Research Scientist, Bureau of Rural Sciences, Canberra, Australia

This timely companion to the critically acclaimed **Encyclopedia of Pest Management** ranges across a broad spectrum of disciplines including botany, ecology, zoology, agriculture, engineering, environment, public health, and soil and water sciences to identify diverse pest species that damage and destroy crops, livestock, and forest products. Containing completely new entries, **Volume II** provides immediate and precise information on a wide spectrum of scientific and human topics, concepts, methodologies, strategies, solutions, questions, and dilemmas

About the editor: David Pimentel is Professor of Insect Ecology and Agricultural Sciences, Department of Entomology and Department of Ecology and Evolutionary Biology, Cornell University, Ithaca, New York. The author or coauthor of over 600 scientific publications including 24 books, Dr. Pimentel has served or is serving with the National Geographic Society Research Committee, the Climate Institute, the International Food Policy Institute, the Royal Swedish Academy of Sciences, and the Chinese Academy of Science, among others. He received the B.S. degree (1948) from the University of Massachusetts, Amherst, and the Ph.D. degree (1951) from Cornell University.

53612



www.crcpress.com



CRC Press

Taylor & Francis Group
an **informa** business

www.taylorandfrancisgroup.com

6000 Broken Sound Parkway, NW
Suite 300, Boca Raton, FL 33487

270 Madison Avenue
New York, NY 10016

2 Park Square, Milton Park
Abingdon, Oxon OX14 4RN, UK