Integrated Pest Management

Concepts of IPM

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"Integrated pest management (IPM) is an ecologically-based pest control strategy that relies heavily on natural mortality factors such as natural enemies and weather, and seeks out control tactics that disrupt these factors as little as possible. IPM uses pesticides, but only after systematic monitoring of pest populations and natural control factors indicate a need. Ideally, an integrated pest management program considers all available pest control actions—including no action—and evaluates the potential interaction among various control tactics, cultural practices, weather, other pests, and the crop to be protected." [1]

While dozens of definitions have been proposed for IPM (see: http://ipmnet.org/IPMdefinitions/), the definition above has been widely accepted by the agricultural community for 30 years. In particular, it points to IPM's ecological foundation and to the importance of monitoring and selection of multiple control practices.

Most of the benefits of implementing IPM are centered on the reduction of pesticide use. Fewer pesticide applications result in savings for materials and application costs. Environmental contamination and worker health problems can also be reduced, and energy for the manufacture and application of pesticides is conserved. Pesticide reduction reduces the development of pesticide resistance and results in stronger natural enemy populations reducing the likelihood of pest outbreaks.

Depending on the complexity of the management system, an IPM program may target a single pest, a pest category (e.g. insects, weeds, diseases or rodents) or the whole pest complex. While traditional pest control considers each pest exclusively, IPM takes into account the interactions among pests, beneficial organisms, the environment, and the crop.

Development of an IPM system requires a thorough understanding of the biology of the crop (or resource) and of the pest complex. The IPM concept was developed from the realization that most pesticide applications affect both pests and beneficial organisms in the crop, sometimes to the disadvantage of the grower.

An IPM system attempts to maintain pest populations below economically damaging levels by using a balance of biological, cultural, chemical, genetic, and other control methods. IPM systems are flexible and programs may vary with time of year, location, and type of crop. Many books, manuals and websites are devoted to discussions of general IPM principles and to the application of IPM to specific agricultural and urban systems. However, the following components are generally found in IPM programs:

1. **Management units** Monitoring is conducted with the aim of providing results for the management of a specific management unit – the part of the system that will receive the same pest control decisions. The unit may be part of a field, a single field or several fields. Chemical control decisions are sometimes based on the area that can be covered by a single spray tank.
2. **Key pests** An IPM program targets specific pests, which may include insects, mites, plant diseases, weeds or vertebrates. In the development of an IPM program, these pests are identified and monitoring and control programs are designed for each of these pests.
3. **Monitoring** Sampling should accurately assess the pest pressure and the abundance of beneficial organisms in the management unit. Monitoring is conducted so that management actions can take place in a timely and effective manner.
4. **Pest action thresholds** Keeping fields entirely pest free is neither necessary nor desirable. Most crops can tolerate low pest infestation levels without any yield loss. IPM seeks to reduce pest numbers below economically damaging levels rather than eliminate infestations. Pesticides should be applied only when economically justified by the numbers of pests present.
5. **Use of multiple controls and tactics** Control tactics should be employed to make the crop less favorable for pest survival and reproduction, while disturbing the rest of the ecosystem as little as possible. Combining different control tactics into an overall strategy balances the strengths of each against any individual weaknesses. Using different techniques also reduces the probability of the development of pest resistance. Control tactics should be compatible with beneficial organisms and the environment.

Developing or implementing an IPM program for a crop involves a systematic application of knowledge about the crop and the pests involved. The following sources may be useful in acquiring and applying that knowledge:

- ♦ Oregon State University Integrated Plant Protection Center—http://www.ipmnet.org/
- ♦ Washington State University Extension IPM—http://www.ipm.wsu.edu/
- ♦ University of Idaho Pest Management Center—http://www.uihome.uidaho.edu/ipm
- ♦ US Environmental Protection Agency: Integrated Pest Management (IPM) Principles—http://www.epa.gov/opp00001/factsheets/ipm.htm
- ♦ Radcliffe's IPM World Textbook—http://ipmworld.umn.edu
- ♦ Flint, M.L, 2012. IPM in Practice, Principles and Methods of Integrated Pest Management. Univ. of Calif. Publ. 3418. 292 pp.

[1] Flint, M. L. and R. Van den Bosch. 1981. Introduction to Integrated Pest Management. Plenum Press. 240 pp.

Biological Control

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Introduction

Biological control (or biocontrol) is a key component in establishing an ecological and integrated approach to pest management. We define biological control as the decline in pest density as a result of the presence of natural enemies. The degree of pest decline might be in the form of partial or complete pest suppression. We use the terms “natural enemies,” “beneficials,” and “biocontrol agents” synonymously to refer to predators, parasites (or parasitoids), and diseases of pests.

Biocontrol is generally more compatible with organic and sustainable agricultural approaches and less compatible with pesticide-dependent agriculture, especially when the less selective, more broad-spectrum chemistries are used. Biocontrol agents tend to be highly susceptible to non-selective pesticides and so pests that are normally controlled by natural enemies can be released from suppression due to short and medium-term pesticide effects. The term “secondary pest outbreaks” is used when this scenario occurs. This reduction in natural enemies can also produce dependence on further pesticide usage and result in a hard-to-break cycle of chemical dependency.

Ideally, natural enemies reproduce on their own and are self-sustaining, are compatible in combination with other integrated control tactics, and are not harmful to other aspects of the ecosystem. Generalist natural enemies (such as most aphid predators) can switch readily among alternative food sources. Thus, when pest numbers are low, the generalist natural enemies may maintain population numbers by consuming other prey species. Specialist natural enemies (such as most parasitoid wasps) depend on one or more food choices and usually increase and decline with the pest population (after a certain lag period). Natural enemies can be disrupted by chemicals, can struggle in poor habitat with low pest numbers, are in some cases difficult to sample or even detect (and thus can be undervalued as to their benefits), and may be incapable by themselves of suppressing pests below damage thresholds.

Insect pests are also susceptible to entomopathogenic nematodes (roundworms) and a variety of diseases caused by pathogens, which include viruses, bacteria, fungi, and protozoa. Natural populations of insect pests are commonly attacked by pathogens, and some pathogens have been mass-produced and are used as biocontrol agents (e.g., microbial insecticides).

Thus, natural enemies, especially a combination of generalists and specialists, can be an extremely useful part of pest management programs that recognize and encourage their activity. At the same time, one must keep in mind that biological control agents can have unanticipated effects that include attacking beneficial and native species. New biocontrol agents increasingly require long-term, stringent evaluations in quarantine for non-target effects and efficacy in controlling the target pest before release. Biocontrol agents that are candidates for introduction may be rejected if, in addition to the target species, they attack native non-pest species. Another risk of introducing new biocontrol agents is the risk of host shifting, which is an unexpected change of attacking the pest (host) despite previous efforts to determine host range.

Types of biological control

In addition to the philosophy of “doing nothing” in order to allow natural biological control to work, there are three principal approaches involving human activity:

1. Classical biological control
2. Augmentative biological control
3. Conservation biological control

Classical biological control

Classical biological control is the importation of natural enemies for release and permanent establishment in a new region. In the Pacific Northwest (PNW), we have had very few cases of highly successful classical biocontrol of insect pests, but there have been many successful classical weed biocontrol cases using insects (see the PNW Weed Management Handbook). One successful insect biocontrol agent, the filbert aphid parasite (Trioxys pallidus), was imported from Europe and introduced (in small numbers) by OSU scientists in the mid-1980s. Since then, this tiny wasp has spread throughout the growing region and generally maintains the filbert aphid below treatment thresholds. In another case, the spread of and damage by the apple ermine moth (Yponomeuta malinellus), has been greatly reduced by the successful introduction of a wasp parasite (Ageniaspis fuscicollis) in the late 1990s. A cooperative biocontrol program among USDA-APHIS, ODA, and OSU for cereal leaf beetle began in 2000 and was considered successful by 2010. The establishment of the larval parasitoid, Tetrastichus julis (Eulophidae), yielded control below thresholds in some regions of the PNW, especially when combined with altered cultural practices (tillage, irrigation, crop rotation, etc.) and pesticide application. In some cases, 100% parasitism was achieved. A small wasp in the family Eulophidae, Colpoclypeus florus, a native of Europe, has been credited as a major biocontrol agent of leafroller pests such as the oblique-banded and pandemis leafrollers in Washington, and has also been collected in Western Oregon. An egg-larval parasitoid, Ascogaster quadridentatus (Braconidae) was introduced to help manage codling moth, a key pest of apple and pear. Economic success of this introduction is unknown, however recovery of this parasitoid from codling moth has been reported.

Previous PNW classical biocontrol efforts included programs directed at Russian wheat aphid, orchard leafrollers, larch casebearer, and cherry bark tortrix. Searches for biological control agents for two new invasive pests—spotted-wing drosophila (SWD, Drosophila suzukii) and brown marmorated stink bug (BMSB, Hyalomorpha halys)—were initiated in 2011. Several wasps were imported from Korea for quarantine, testing, and potential release against SWD. The BMSB egg parasitioid, Trissolcus japonicus, was found established outdoors in Vancouver, Washington in 2015 and Portland, Oregon in 2016 and has been detected yearly in both areas. As a result of this finding, experimental releases of the parasitoid have begun in Oregon.

Augmentative biological control

Augmentative or supplemental biological control typically involves the mass-production and repeated releases of natural enemies. This approach is used most often for slow-moving pests such as mites and aphids, in enclosed spaces such as greenhouses, by home gardeners, and in organic agriculture where few disruptive chemicals are used. The dispersal capability of the natural enemy should be taken into consideration when matching a natural enemy for control of the pest. For example, many homeowners have wasted money using ladybug adults to control aphids, only to see them disperse away within minutes. If biocontrol agents are native or established from exotic sources, then a release can be directed to augment and improve the rate of natural colonization and control. If the biocontrol agent is non-native and overwintering success is not expected, only in-season benefits will occur. It was demonstrated in Oregon strawberries during the mid-1990s that the PNW-native predatory mite Neoseiulus fallacis can be purchased from insectaries and released in the early fall to re-establish healthy populations that normally control the twospotted spider mite, in cases where pesticides had previously been used to control root weevils (Croft and Coop 1998). Some other predator mite species, such as N. californicus, which may be more readily available for purchase than is N. fallacis, may provide in-season control but may not survive the relatively colder winters that N. fallacis is adapted to survive. Since natural enemies are all specialized to some degree, it's important to know the pest and which agent(s) are appropriate for the given situation. Table 1 lists some target pests commonly found in home garden and agricultural systems and the associated commercially-available beneficial organisms. Steps for acquisition and release of biocontrol agents must be planned carefully and followed. Release guidelines depend on an understanding of the biology of the pest, the natural enemy, and the influence of the host plant on both. Conservation efforts (below) can in some cases enhance the outcome of augmented biocontrol agents.

Conservation biological control

Conservation biological control refers to the manipulation and/or protection of habitat and resources to support and encourage natural enemies in order to increase their numbers and effectiveness. This may include the use and encouragement of the natural enemies' needs, such as nectar and pollen, alternative hosts, and certain types of non-disrupted habitat. These resources all can potentially enhance the fecundity, longevity, and survival of natural enemies.

Some tactics for conservation biological control include:

- ♦ Careful use of pesticides and tillage to avoid disturbing natural enemies. There are secondary pests that only reach economic pest levels when their natural enemies are disrupted by pesticides, that were applied in order to control a different species. Using least toxic and selective controls in the place of broad-spectrum compounds (such as most organophosphates, carbamates, and pyrethroids) can help prevent secondary pest outbreaks. Online databases and listing of pesticide effects on beneficial organisms include:
  - http://enhancedbc.tfrec.wsu.edu/opened/
  - http://ipm.ucanr.edu/PMG/r302900111.html
  - http://www.intermountainfruit.org/pesticide-tables/toxicity-pollinators
- ♦ Non-crop plantings in or around the crop field that may provide shelter, alternative prey, nectar, and pollen. Table 2 gives some examples of flowering plants that are visited by natural enemies.
- ♦ Food sprays (such as yeast and sugar sprays) to attract parasitoid wasps, lady beetles, lacewings, and hoverflies.
- ♦ Manipulating crop and non-crop architecture in ways that improve natural enemy activities (for example, using wind-break plantings as a barrier to prevent dry, dusty conditions favorable to pest mite flare-ups. Predatory mites need sufficient humidity and can be inhibited by such conditions.)

The effects of the above tactics are not well understood, and they can be less consistent than other forms of biological control due to the complex interactions involved. However, they do make use of the local natural beneficial species already present in the landscape. Note also that conservation biological control efforts can enhance natural enemies released in classical and augmentative biological control programs. For example, some of the most commonly used methods for providing floral resources (e.g. pollen, nectar, nectaries), also known as beneficial "insectary plantings," include:

1. Planting within the crop field in strips or smaller blocks
2. Using perennial and annual border plantings
3. Planting within hedgerows
4. Establishing cover crops
5. Careful management of flowering weeds

Coincidentally, these insectary planting methods also can provide habitat and alternate hosts for natural enemies in certain situations. Shelter and alternate hosts also can be supported through methods such as careful rotation, alternate row harvest, and "beetle banks," which are graded low banks of dense grasses placed within the field or in fence row corridors inhabited by appropriate vegetation.

Just as when selecting any new crop management method, choosing insectary plantings for conservation biological control should include consideration of numerous biological, agronomic, and economic factors. Table 3 gives an example of the range of factors to consider in designing an insectary planting. To justify the continued use of an insectary planting, the on-site assessment should consider the same factors as the preliminary selection process, as well as a sampling of pests and beneficials within and surrounding the crop.

Considerations for incorporating insectary plantings to sustain natural enemies

Timing of flowering

1. Will the floral resources be present when needed?
2. Will the flowers attract natural enemies to or away from the target pest at certain times?

Characteristics of the natural enemies

1. What are the relative preferences that key natural enemy and pest species have for the different flowers?
2. What are the different requirements for nectar, pollen, shelter, and alternate host food among these key species?
3. What are the relative foraging ranges and dispersal abilities of these key species?

Agronomic considerations

1. How competitive are the plantings with the crop or other weeds?
2. Do the plantings have the potential to harbor weeds or be weeds themselves?
3. Can the plantings serve as an alternate host for crop disease?
4. Are the plants toxic to any livestock or other local animals?

Economic and management considerations

1. Can the planting be harvested as an additional crop?
2. What are the costs of seed, establishment, and maintenance?
3. How do these costs compare to other management options?
4. Are the plantings compatible with the main pest management plan?

Resources for implementation of biological control

The IPM Practitioner's 2015 Directory of Least Toxic Pest Control Products. A regularly updated, comprehensive, statewide listing of biological control agents and other "least toxic" pest control products for a variety of agricultural, urban, and domestic uses, and their producers and distributors. Bio-Integral Resource Center http://www.birc.org/Directory.htm

An informative 12 minute video on habitat and biological control made in 2015 by Eric Brennan, PhD researcher at USDA in Salinas, CA, that features the article by Dan Karp, "Co-managing fresh produce for nature conservation and food safety," recently published by the National Academy of Sciences: Biological Control Buffet in the Salad Bowl of America https://www.youtube.com/watch?v=zLvJLHERYJI

Sandhu, H. S. Wratten, R. Costanza, J. Pretty, J. R. Porter, and J. Reganold. 2015. Significance and value of non-traded ecosystem services on farmland. PeerJ 3:e762; DOI 10.7717/peerj.762 https://peerj.com/articles/762.pdf

Oregon Department of Agriculture provides a list of invertebrates approved for importation into Oregon. Except as otherwise provided in rules of the ODA, invertebrate species listed in this list may be imported, possessed, sold, purchased, exchanged or transported within the state without an ODA permit. A permit for the importation, possession, or intrastate transportation of ODA-approved species may be required by the US Department of Agriculture, Animal and Plant Health Inspection Service, Plant Protection and Quarantine, Form 526. https://www.oregon.gov/ODA/shared/Documents/Publications/IPPM/OregonApprovedInvertebrateList.pdf

The Xerces Society. A nonprofit organization formed in 1971 which protects wildlife through the conservation of invertebrates and their habitat. Their focus has expanded beyond native pollinators to include all invertebrates including other native species, predators, and parasitoids. They have programs to document the impacts of pesticides on invertebrates including biocontrol agents. Xerces has resources to provide education and training on conservation biological control and are very active in the Pacific Northwest. 628 NE Broadway Ste 200, Portland OR 97232 USA; tel: 855-232-6639 https://www.xerces.org/

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Table 1. Target pests and beneficial organisms often used for augmentative biological control releases

Aphid (See also soft-bodied arthropods)	predatory midge	*Aphidoletes aphidimyza*
	parasitoid wasp	*Aphidius ervi, A. matricariae, A. colemani, Lysiphlebus testaceipes, Trioxys pallidus*
	big-eyed bug	*Geocoris pallens*
	lady beetle ("ladybug")	*Hippodamia convergens*
	lacewing	*Chrysoperla downesi, C. plorabunda, C. rufilabris*
	minute pirate bug	*Orius insidiosus, O. minutus, O. tristicolor*
Armyworm (See also Butterfly and moth)	braconid parasitoid wasp	*Chelonus insularis*
Black fly larvae	bacterial endotoxin (Bti)	*Bacillus thuringiensis* var. *israelensis* (e.g., Bactimos, Teknar, Vectobac)
Butterfly and moth larvae and eggs of beetle pests in stored grain products, such as almond moth, Indian meal moth, grain weevil	parasitoid wasp	*Bracon hebeter*
Butterfly and moth eggs and young larvae: beet armyworm, cabbage looper, corn earworm, cutworm, diamondback moth, imported cabbageworm, codling moth and other orchard moths, tobacco budworm	viral pathogen	Nuclear polyhedrosis virus (NPV)
	bacterial endotoxin (Btk, Bta)	*Bacillus thuringiensis* var. *kurstaki* (e.g., Dipel, Javelin, Attack, Thuricide, Bactospeine, Safer's Caterpillar Killer), *Bacillus thuringiensis* var. *aizawai* (e.g., Certan)
	parasitoid wasps of eggs	*Trichogramma minutum, T. pretiosum, T. platneri*
Codling moth larvae	granulosis virus pathogen	*Baculovirus carpocapsae*
Flea	parasitic nematode	*Steinernema carpocapsae, S. feltiae*
Fly (garbage- and manure-breeding)	parasitoids of puparia	*Melittobia digitata, Muscidifurax raptor, Muscidifurax zaraptor, Nasonia vitripennis, Pachcrepoideus vindemiae, Spalangia cameroni, S. endius*
	histerid beetle predator	*Carcinops pumilio*
Fungus gnat (larvae)	predatory mite	*Hypoaspis miles, H. aculeifer*
	parasitic nematode	*Heterorhabditis megidis, Steinernema carpocapsae, S. feltiae*
	bacterial endotoxin (Bti)	*Bacillus thuringiensis* var. *israelensis*
Grasshopper (nymphs and adults)	protozoan	*Nosema locustae*
Larvae and grubs that pupate in the soil: cucumber beetles, dampwood termites, flea beetles, root weevils, wireworms	parasitic nematodes of larvae	*Heterorhabditis bacteriophora, H. heliothidis, H. megidis, Steinernema feltia, S. carpocapsae, S. riobravis*
Leafminer	braconid parasitoid of larvae	*Dacnusa sibirica*
Mealybug	lady beetle ("mealybug destroyer")	*Cryptolaemus montrouzieri*
Mite: twospotted spider *(Tetranychus urticae)*	predatory mite	*Amblyseius hibisci, A. mckenziei, Galendromus occidentalis, Mesoseiulus longipes, Neoseiulus californicus, N. fallacis, Phytoseiulus persimilis, P. macropilis*
	predatory six-spotted thrips	*Scolothrips sexmaculatus*
	minute pirate bug	*Orius minutus, O. tristicolor*
	big-eyed bug	*Geocoris pallens*
Mosquito larvae	predatory fish	*Gambusia affinis* spp.(only in manmade water bodies or containers that have no connection to natural waterways)
	bacterial endotoxin (Bti)	*Bacillus thuringiensis* var. *israelensis* (e.g., Dunks, Bactimos, Vectobac, Teknar)

Table 1. Target pests and beneficial organisms often used for augmentative biological control releases

Scale: armored scale, oleander scale, San Jose scale, ivy scale	lady beetle	*Chilocorus fraternus*
Soft scale: citrus black scale, black/brown hemispherical, nigra scale (See also soft-bodied arthropods)	parasitoid wasp	*Metaphycus helvolus*
Soft-bodied arthropods: thrips, scale, aphid, spider mite, whitefly, eggs of harmful pests	lacewing larvae (in larval stage)	*Chrysoperla downesi, C. plorabunda, C. rufilabris*
	fungal pathogen	*Beauveria bassiana*
	lady beetle	*Chilocorus fraternus, Hippodamia convergens*
	pirate bug	*Orius minutis, O. tristicolor*
	predatory thrips	*Scolothrips sexmaculatus*
Thrips larvae (*See also* soft-bodied arthropods)	predatory mite	*Amblyseius cucumeris, A. mckenziei, A. barkeri, A. degenerens*
	lacewing	*Chrysoperla downesi, C. plorabunda, C. rufilabris*
	minute pirate bug	*Orius minutus, O. tristicolor*
Wax moth larvae (in honeycombs)	bacterial endotoxin (Bta)	*Bacillus thuringiensis* var. *aizawai* (e.g. Certan)
Weevil in landscape plants	parasitoid wasps of larvae	*Anisopteromalus calandrae*
	parasitic nematode	*Heterorhabditis heliothidis, H. medidis, Steinernema carpocapsae, S. feltiae, S. riobravis*
Whitefly nymph (*See also* soft-bodied arthropods)	parasitoid wasps of eggs	*Encarsia formosa, Eretmocerus californicus*

1 Lady beetles include many species in the family Coccinellidae, order Coleoptera.

2 Lacewings include many species in the families Chrysopidae and Hemerobiidae, order Neuroptera.

3 Parasitoid and predatory wasps include a large number of species in families such as Aphelinidae, Aphidiidae, Braconidae, Chalcididae, Crabronidae, Encyrtidae, Eulophidae, Ichneumonidae, Mymaridae, Pompilidae, Pteromalidae, Scelionidae, Specidae, and Trichogrammatidae, order Hymenoptera.

4 Hoverflies include many species in the family Syrphidae, order Diptera.

5 Predatory bugs include many species in families such as Anthocoridae, Lygaeidae, Nabidae, Pentatomidae, and Reduviidae, order Heteroptera.

6 Minute pirate bugs include many species in the family Anthocoridae, order Heteroptera.

7 Big-eyed bugs include many species in the family Lygaeidae, order Heteroptera.

8 Parasitoid Tachinid flies include many species in the family Tachinidae, order Diptera.

9 Bees include many species in families such as Anthophoridae, Apidae, Halictidae, Andrenidae, Colletidae, and Megachilidae, order Hymenoptera.

Table 2. Flowering plants visited by beneficial insects that can aid biological control conservation efforts

Common name (botanical name)	Beneficial insects
Apiaceae (Carrot family)	
Angelica *(Angelica)*	lady beetle ("ladybugs"), lacewing
Anise *(Pimpinella anisum)*	parasitoid wasp
Blue lace *(Trachymene caerulea)*	parasitoid wasp
Caraway *(Carum caryi)*	hoverfly, minute pirate bug and big-eyed bug, lacewing, parasitoid wasp
Chervil *(Anthriscus cerefolium)*	parasitoid wasp
Coriander *(Coriandrum sativum)*	hoverfly, parasitoid wasp, parasitoid tachinid fly
Dill *(Anethum graveolens)*	hoverfly, lady beetle, parasitoid wasp
Fennel *(Foeniculum vulgare)*	hoverfly, parasitoid wasp, parasitoid tachinid fly
Lovage *(Lovisticum officinale)*	parasitoid wasp
White lace flower *(Ammi majus)*	hoverfly, predatory bug, lady beetle, parasitoid wasp, parasitoid tachinid fly
Wild carrot *(Daucus carota)*	hoverfly, predatory bug, lady beetle, lacewing, parasitoid wasp
Asteraceae (Daisy family)	
Blazing star, gayfeather (*Liatrus* spp.)	minute pirate bug, big-eyed bug, parasitoid wasp
Chamomile *(Anthemis nobilis)*	lady beetle
Cosmos *(Cosmos bipinnatus)*	hoverfly, lacewing, minute pirate bug
Golden marguerite *(Anthemis tinctoria)*	lady beetle, parasitoid wasp, parasitoid tachinid fly
Goldenrod *(Solidago altissima)*	soldier beetle, predatory bug, lady beetle, parasitoid wasp
Marigolds, signet *(Tagetes tenuifolia)*	minute pirate bug, parasitoid wasp
Mexican sunflower *(Tithonia tagetifolia)*	hoverfly, minute pirate bug
Sunflower *(Helianthus annuus* and *H. debilis)*	hoverfly, lady beetle, parasitoid wasp
Tansy *(Tanecetum)*	hoverfly, lady beetle larvae, parasitoid wasp
Yarrow, milfoil *(Achillea millefolium)*	hoverfly, parasitoid wasp
Yarrows *(A. macrophylla, A. taygetea,* etc.*)*	hoverfly, parasitoid wasp
Brassicaceae (Cabbage family)	
Broccoli *(Brassica oleracea)*	hoverfly, parasitoid wasp
Sweet alyssum *(Lobularia maritima)*	hoverfly, parasitoid wasp, parasitoid tachinid fly
Globe candytuft *(Iberis umbellata)*	hoverfly
Mustards *(Brassica hirta* and *B. juncea)*	hoverfly, minute pirate bug, big-eyed bug
Dipsaceae (Scabiosa family)	
Cephalaria *(Cephalaria giganitica)*	hoverfly, parasitoid wasp
Dipsacus *(Dipsacus* spp.*)*	hoverfly
Pincushion flower *(Scabiosa caucasica)*	hoverfly, parasitoid wasp
Scabiosa *(Scabiosa atropurpurea)*	hoverfly
Fabaceae (Legume family)	
Alfalfa *(Medicago sativa)*	bee, predatory bug, lacewing, lady beetle, parasitoid wasp
Clover *(Trifolium* spp.*)*	bee, predatory bug, lacewing, lady beetle
Vetch *(Vicia* spp.*)*	bee, predatory bug, lacewing, lady beetle
Hydrophyllaceae (Waterleaf family)	
Fiddleneck/Phacelia *(Phacelia tanacetifolia)*	bee, predatory bug, hoverfly
Lamiaceae (Mint family)	
Germander *(Teucrium* spp.*)*	bee, parasitoid wasp
Polygonaceae (Buckwheat family)	
Buckwheat *(Eriogonum* spp. and *Fagopyrum* spp.*)*	hoverfly

See notes for Table 1.

Entomopathogenic Nematodes

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Insect-pathogenic, or entomopathogenic nematodes, are a group of soil-dwelling roundworms which only kill insects that live in, on, or near the soil surface, usually closely associated with plants. These nematodes can occur naturally in soil and are found in most places where plants grow. Research has demonstrated that entomopathogenic nematodes can be mass produced, have a narrow host specificity against pests, and are safe to plants and vertebrates; and, therefore, the U.S. Environmental Protection Agency has exempted them from all registration requirements and related regulation. Entomopathogenic nematodes have been available commercially to agriculturists for several years and have been used in a variety of cropping systems.

There are two main groups of entomopathogenic nematodes: the steinernematids and the heterorhabditids. Both have similar life cycles, and only the free-living, infective juvenile stage is able to infect the target (pest) insect. It is the juvenile stage that is found in or on the soil, searching out a host to infect. In fact, the juvenile form is the only form found outside of the host.

Slug-pathogenic, or malacopathogenic nematodes, are also a group of soil-dwelling roundworms, a novel method and approach that has been used in Europe to kill slugs with some success. One of the most widely established, commercially-available slug biocontrol agents in Europe is the nematode Phasmarhabditis hermaphrodita (Schneider). This nematode has been shown to be associated symbiotically with a bacterium that uses an endotoxin that kills slugs. The nematode locates slugs in the soil and enters the slug's mantle cavity. After the slug dies, the nematodes multiply over the decaying slug body and then migrate back into the soil where infect more slugs, if conditions are favorable. Parasitic nematodes were recently found in CA, but are currently restricted in this country due to biosecurity reasons, so are unavailable for purchase. Research focusing on discovering and testing pathogenic nematodes in PNW fields will prove to be valuable for use as a biological control agent for control of slugs.

This document focuses on insect-pathogenic nematodes.

Nematode selection for Insects

The choice of an entomopathogenic nematode *(Steinernema* spp. or *Heterorhabditis* spp.*)* depends on the targeted insect pest. In general, nematodes in the genus Steinernema are considered "sit-and-wait predators" or ambushers and are used against insects whose immature stages (larvae or pupae) spend most of their time at or near the soil surface. Other species are highly mobile and roam through the soil searching for potential hosts. The host-finding strategy of most *steinernema* is to wait until the prey bumps into the nematode, and then infects it. In contrast, nematodes in the genus *Heterorhabditis* actively seek out or hunt for their prey, sometimes several inches below the soil surface, and stay in one spot for an extended period of time. Thus, nematodes in the genus *Steinernema (S. feltiae)* are the best choice against fungus gnat larvae, often found on the soil surface of potted plants, while the genus *Heterorhabditis, (H. megidis, H. marelatus,* or *H. bacteriophora)* are the best choices against the black vine weevil, deeper in the soil. Recently, H. bacteriophora and S. feltiae appeared promising for targeting larvae of spotted wing drosophila. There are over ten entomopathogenic nematodes commercially-produced as a biological insecticide for over 25 insect pests.

There is some overlap between the various species with regards to host-finding ability. Consult a nematode manufacturer/supplier for selection of the proper entomopathogenic nematode product.

Life cycle

An infective juvenile may at first move randomly, and then find their insect host via carbon dioxide, host odor or damaged plant odors. Once a juvenile locates an insect, the juvenile enters via a natural opening; or, in certain instances, it may penetrate a weak spot in the insect's cuticle. Insect larvae and pupae are more susceptible to nematodes since adult insects are often more mobile. Once inside the host's blood system, the juvenile releases a symbiotic bacterium that it carries. The bacteria are released into the blood of the host, rapidly multiply, and produce compounds that kill the host insect generally within 48 hours. The bacteria protect and preserve (via antibiotics) the dead insect from invasion by unwanted, contaminating soil microbes and the nematodes provide shelter for the bacteria. The infective nematodes complete one to several generations inside the host, feeding on the bacteria and nutrients within the dying host. Only when all the host tissues have been consumed does a new generation of juveniles emerge, all carrying the symbiotic bacteria with them in search of new hosts (see Figure 1).

One generation from egg to egg typically takes from 4 to 7 days. In most instances, there are at least two generations inside a host before the new juveniles emerge seeking a new host, so from the time of first infection by juveniles to the time "new" juveniles emerge may be from 8 to 14 days. The length of time is determined by the temperature of the soil, the size of the host, and which nematode is involved. A large host such as a cutworm will support several generations before conditions become too "crowded" and juveniles emerge, compared to a strawberry root weevil larva, where there may be only one or two generations before juvenile emergence. Similarly, a large nematode such as *S. carpocapsae* has fewer generations than *S. feltiae* when infecting similar-size hosts.

Application methods

Though the adult stage of some insect pests also is susceptible, entomopathogenic nematodes generally are used for controlling the soil-borne larval or pupal stages of a pest. Therefore, entomopathogenic nematodes most often are applied by drench or band application. While broadcast application has been used at times, the immature pest insect usually is not located between the crop rows as there is usually no food source there. If, however, the crop has a closed canopy like cranberries or mint, a broadcast application may be warranted. An adjuvant may help. Select your application method wisely, as it may impact greatly the success of host location, infection, and control by the entomopathogenic nematodes.

Entomopathogenic nematodes come in a variety of formulations: water-dispersible granules, nematodes on gel, micronized vermiculite, nematode wool, and an aqueous suspension of nematodes. These formulations are intended to be mixed with water to release the nematodes through common application equipment such as small pressurized sprayers, mist blowers, electrostatic sprayers, or even helicopters (aerial application). Some more promising methods for applying entomopathogenic nematodes are emerging. One uses irrigation systems in a manner similar to chemigation. Another uses nematode-filled capsules which include attractants or feeding stimulants for the pest; this draws in the pest for infection rather than relying on the nematode to find the pest.

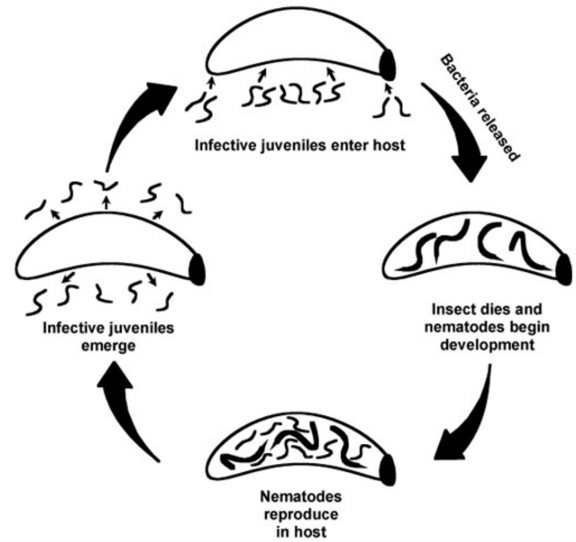
Regardless of the method, nematodes can withstand application pressures of approximately 300 psi and can pass through most spray nozzles without difficulty, though operating pressures between 20 to 60 psi generally are sufficient. Keep in mind that nozzle orifices should not be smaller than 50 microns (0.00019685 inch), and that any screens in the system should have an opening of at least 50 mesh (0.0117 inch) or larger to allow the free passage of nematodes through the system. In any case, follow the manufacturer's directions.

Nematodes require a film of water around soil particles to move through the soil profile in search of a host. Therefore, pre-irrigate the soil in the treatment area with about 0.25 to 0.5 inch of water no later than a few hours before application of the nematodes. Following the application, "water in" the nematodes with an additional 0.5 inch of water to wash them off of foliage and protect them from damaging UV radiation. Further irrigation to maintain adequate soil moisture for at least 7 days following nematode application also is recommended. Be careful not to over-irrigate, because excess water inhibits the movement of oxygen in the soil, and the nematodes will drown. A good rule of thumb is to avoid standing water in your fields.

Key points for success in using entomophathogenic nematodes

- When applying agrichemicals in the area where entomopathogenic nematodes are to used, be sure that there is enough separation time between applications of toxic compounds and entomopathogenic nematodes (Table 1). Some chemicals have been found to affect nematode efficacy when nematodes are exposed to them. These should be applied with care when used in conjunction with nematodes.
- Entomopathogenic nematodes require a moist, not saturated, soil environment so they can move around and locate their host.
- Soil temperature where nematodes are to be applied should be above 55°F and less than 90°F. Nematodes are also affected by suboptimal soil type, thatch depth, and irrigation frequency.
- Protect nematodes from excessive exposure to ultra violet (UV) rays which can inactivate and kill them.
- Time application of entomopathogenic nematodes to target the susceptible stage of the pest.
- Select the proper nematode species to match the most susceptible pest stage.
- Storage of formulated nematode species varies: Steinernematids at 39-46°F; Heterohabditids at 50-60°F. Do not leave in a hot vehicle.
- Select the application rate and method to maximize contact between entomopathogenic nematodes and the target pest.
- In all cases, refer to the manufacturer's label for recommendations.

Note: We appreciate the contributions of past employees of Oregon State University, Peter Guthro and Ralph Berry, to this document.



ENTOMOPATHOGENIC NEMATODE LIFE CYCLE

Fig. 1. Generalized life cycle of a steinernematid nematode. Reprinted with permission from Shapiro-Ilan, D.I. and Gaugler, R. (n.d.) Nematodes. In Biological Control: A Guide to Natural Enemies in North America (Anthony Shelton, editor) — http://www.biocontrol.entomology.cornell.edu//pathogens/nematodes.html

Table 1. Chemical-use patterns with nematodes[1]

Compiled from D. Shetlar, 1999, and K. Smith, 1999; Alumai and Grewal, 2004

May be tank-mixed with nematodes			
Compound	**Trade name**	**Compound**	**Trade name**
acephate	Orthene	fenoxycarb	Logic
azadirachtin[2]	Azatin, Neem[2]	fertilizers	various
Bacillus thuringiensis (Bt)	M-One, Dipel	fipronil[2]	Chipco Choice[2]
benomyl	Benelate	glyphosate	Roundup
bifenthrin	Talstar	insecticidal soap[2]	various
bromine-chlorine	Agribrom	iprodione	Chipco 26019
carbaryl[2]	Sevin[2]	isofenphos	Oftanol
carbofuran[2]	Furadan[2]	kinoprene	Enstar
chlorothalonil	Daconil	metalaxyl	Subdue
chlorthal dimethyl	Dacthal	methidathion	Supracide
copper hydroxide	Kocide	methomyl[2]	Lannate[2]
cyfluthrin	Tempo	methoprene	Apex
cythion	Malathion	oryzalin	Surflan
diazinon	Knox-out	oxamyl[2]	Vydate[2]
dienochlor	Pentac	oxazoidinedione	Ornalin
diflubenzurion	Dimilin	pentachloronitrobenzene	Terrachlor
endosulfan	Thiodan	thiophanate-methyl	Zyban
esfenvalerate	Asana	triademefon	Bayleton
etridiazole	Terrazole		

1-wk separation		2-wk separation	
anilazine	Dyrene	ethoprop	Mocap
bendiocarb	Turcam, Ficam	fenamiphos	Nemacur
chlorpyrifos	Dursban	isazophos	Triumph
dimethyl benzyl ammonium chloride	Physan 20		
fenarimol	Rubigan		
mercurous chloride	Calo-Clor		
2,4-D	Various		
triclopyr	Turflon, Confront		

[1] Note with caution: Environmental conditions could affect the pathogenicity, virulence and effectiveness of entomopathogenic nematodes with selected pesticide formulations; and moreover the nematode species can differ in their susceptibility and sensitivity to different formulations to the same chemical pesticides

[2] Use pattern not well-established. Monitor closely.

References for Table 1

Alumai, A. and P. Grewal, 2004. Tank-mix compatibility of the entomopathogenic nematodes, *Heterorhabditis bacteriophora* and *Steinernema carpocapsae*, with selected chemical pesticides used in turfgrass. 2004. Biocontrol Science and Technology, 14(7). DOI: 10.1080/09583150410001724334.

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Smith, K. 1999. "Factors Affecting Efficacy," in Proceedings of Workshop—Optimal Use of Insecticidal Nematodes in Pest Management, Aug. 28–30, 1999. S. Polavarapu, ed.

Additional information on entomopathogenic nematodes and their application can be found in:

Labaude, S., and C.T. Griffin. 2018. Transmission success of entomopathogenic nematodes used in pest control. Insects 9, 72, https://doi.org/10.3390/insects9020072

Miles, C., C. Blethen, R. Gaugler, D. Shapiro-Ilan and T. Murray. 2012. *Using entomopathogenic nematodes for crop insect pest control*. PNW Extension Publication 544. http://cru.cahe.wsu.edu/CEPublications/PNW544/PNW544.pdf

Biology and Control of the Garden Symphylan

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Introduction

Garden symphylans *(Scutigerella immaculata Newport)* (GS) are small, white, centipede-like soil arthropods which infest many home gardens and agricultural soils in western Oregon and Washington. They feed on sprouting seeds, roots, and other organic material such as decaying plants and fungi. Economic damage occurs from direct feeding on roots, rhizomes, and tubers from establishment through plant maturity. Seedling death, poor growth, stunted plants, reduced vigor, and yield reduction result. Chronic feeding on the roots of both annual and perennial plants reduces a plant's ability to acquire water and nutrients. This results in a poor root system that manifests as general stunting and distortion of plants as well as increased susceptibility to plant pathogens. Sampling and control of GS is complicated by daily and seasonal vertical movement in the soil which is influenced by soil moisture, temperature, time of day, season, crop stage, and their internally originating feeding cycles.

Selection of appropriate tactics to manage GS is largely determined by the cropping system (no-till versus tillage), soil type and structure, and availability and use of soil applied insecticides.

Conventional growers, organic growers, and small scale gardeners often approach symphylan management from different perspectives, primarily due to economic and scale-dependent factors. However, in all systems, effective management stems from accurate identification of GS and the damage they cause, a general knowledge of their ecology, as well as appropriate sampling methods and control strategies. Correct diagnosis of a GS problem is sometimes tricky, since damage may be exhibited in a number of forms and GS are not always easy to find when damage is observed.

Identification

Garden symphylans are not insects, but members of the class Symphyla. Several species occur in Oregon, but the GS *(S. immaculata)* is the primary species that causes crop damage in the U.S. GS are by far the most common symphyla species found in Oregon agricultural systems.

Garden symphylans are small, whitish "centipede-like" fast-moving creatures that measure about 0.25 inch long when mature. They have 6 to 12 pairs of legs (depending on age) which make them easy to differentiate from common soil insects which only have 3 pairs of legs. Though their color may vary depending on what they have eaten, they are generally whiter and smaller than true centipedes, which are also soil arthropods with many pairs of legs (one pair per body segment) and make quick movements. Millipedes are generally slower moving soil arthropods, with two pairs of legs on each body segment.

Garden symphylan biology

Eggs, immatures, and adults can be found together throughout most of the year. Temperature plays a key role in regulating oviposition, and the greatest numbers of eggs are most commonly deposited in the spring and fall. Eggs are pearly white to tan, spherical with hexagonal shaped ridges, and laid in clusters. Egg incubation period is from 25 to 40 days under typical spring soil temperatures in western Oregon. First instars emerge from the egg with six pairs of legs. Newly hatched GS resemble springtails. The GS has an exoskeleton and, like an insect, sheds it (molts) periodically to grow and enlarge body size. Each of the six subsequent molts results in the addition of a pair of legs and antennal segments. Total time from egg to sexually mature adult (seventh instar) is about 2 to 3 months during typical spring soil temperatures in western Oregon. Two complete generations per year can occur.

Occurrence and movement

Garden symphylans are generally a problem in irrigated crops grown on alluvial soils with very good soil structure. Within these soils, GS tend to occur in "hotspots" encompassing a few square feet to several acres. Hotspots often remain consistent from year to year with little change in populations and only minor lateral spread.

Within a favorable soil habitat GS can migrate from the soil surface to a depth of over 3 feet. The soil profile, structure, composition and water holding capacity determines the depth to which GS migrate. Vertical migration is primarily related to interactions among moisture, temperature, crop stage and endogenous feeding cycles. A general understanding of these interactions is important both for timing and interpreting sampling efforts, and for selecting management tactics.

Garden symphylans tend to aggregate in the top 6 inches of soil when the soil is moist and warm in the spring and fall and tend to form a circular pattern covering a few feet to a number of acres. They move to deeper soil strata during July and August, though can stay at the surface if sufficient moisture is present and no plants are growing. Garden symphylans migrate to the root zone to feed, then return to the deeper strata to molt, evidenced by the large number of molted skins that may be observed in these strata. Since migration is not entirely synchronized within a population, GS are usually present throughout the habitable portion of the soil profile. Presence of GS in the surface soil may also be influenced by other variables that impede movement, such as tillage and compaction from heavy objects (such as tractor tires).

Sampling

Many of the difficulties in effectively managing GS result from unknowns concerning the density and location of populations in a field. Sampling, although often time-consuming, can provide information critical to managing populations effectively. For annual crops, sampling is commonly conducted in April, May, or June, prior to planting. In general, the later in the spring that sampling occurs, the more GS will be found in the soil. Samples that include crop or weed roots generally contain more root-feeding GS than those taken in bare soil. The type and extent of sampling may vary depending on the site conditions (e.g., vegetation, size of area, cropping history), and whether populations have been historically problematic in certain areas of a site.

Three main sampling methods are used: baiting methods, soil sampling methods, and indirect sampling methods. Each method has benefits and drawbacks, and the selection of a sampling method will vary depending on the objectives of the sampling (e.g., detection vs. precise population density estimation), time of year, and site conditions.

Part of the difficulty in sampling is a result of the patchy spatial distribution of GS populations. It is important to be aware that an individual sample unit count provides information about a local region within which that sample unit was taken. Counts will often vary from zero to more than 50 GS per sample unit (i.e., soil core or bait). To obtain information about the spatial patterns of the population, sample units are often taken in a grid pattern. Areas with different cropping histories are generally sampled independently.

In most cases, sampling only measures the density of GS in the surface soil. Therefore, sampling should only be conducted when GS are within this region. The best sampling conditions are, generally, when the soil is warm and moist. Sampling within 3 weeks after major tillage, such as disking, plowing, or spading may not reflect the true population because GS often have not had ample time to reestablish in the surface soil.

To detect or identify a GS problem in a crop, bait for GS in suspected areas within 3 weeks of planting. To sample seedlings or established plants, dig them up in the early morning when GS are close to the soil surface. Inspect their roots and the soil around the roots. They may also be present in roots of grassy weeds in the area.

Soil sampling is the standard/historic method for estimating how many GS are in a field (i.e., approximate number of GS/unit of soil, or population density estimate). Sample unit sizes vary; the most common soil sample units are 6 x 6 x 12 inches (length, width, depth) or cores of 2.5 inches in diameter by 6- to 12-inch depth. When soil samples are taken, the soil from each sample unit is usually placed on a dark piece of plastic or cloth where the aggregates are broken apart and the GS are counted. Sampling is usually conducted when GS are present in the top 6 to 12 inches of the root zone.

Bait samples are generally much faster to take than soil samples, but they are also more variable and more sensitive to factors such as soil moisture, temperature, and presence of vegetation. To bait sample, the soil surface is first lightly skimmed to expose moist soil and one-half of a sliced potato is placed on the soil surface and sheltered with a protective cover (e.g., white pot or 4-inch PVC cap). Baits are generally checked one to three days after placement. Baits are checked by lifting the bait and counting first the GS on the soil, and secondly the GS on the bait. During warm and/or dry conditions, baits are generally checked one to two days after placement as counts decrease if baits are left out for multiple days. In cooler conditions, baits are commonly left out for three to five days. Bait sampling works very well for some applications, though it cannot be used under all conditions. Baiting works best at least two to three weeks after tillage, when the soil has stabilized but before plants are well established. When baiting works well it is a very useful tool, but numerous factors influence this method. Therefore, soil samples should always be taken along with baits in order to confirm the presence/absence of GS.

Plant growth can sometimes be a useful indirect measure of GS populations and is often a good starting point for assessing GS populations. Indirect measures, however, should never be used without some direct sampling to confirm the presence of GS.

Determining the number of samples

Sampling requirements will often vary by site, depending on factors such as cropping history and time of year. Sampling involves establishing a balance between the need to be confident about estimates of the number of GS present (implying a large number of samples) and not investing excessive time and energy into the sampling endeavor (implying a small number of samples).

Follow these guidelines for determining the sample size:

1. Sampling for low population densities (e.g., early in the spring or of highly susceptible crops) requires a greater number of sample units (e.g., 100+) than sampling for high population density (e.g., 30 GS/foot) as smaller population clusters are more difficult to detect. Ten samples may be enough to confirm that a high population density exists.
2. As the variability of the sampling method increases, so does the number of sample units required. Since the baiting method is more variable than the soil sampling method, two to three times more bait than soil sample units are required.
3. For estimation of "economic" population densities in moderately susceptible crops, at least 35 soil sample units, or at least 50 bait units, are commonly used. Depending on the size of the field, and the time of year, considerably more sample units are sometimes used.

Action thresholds

Management decisions, such as those regarding pesticide applications and the intensity of tillage, are sometimes made based on pre-plant GS population density estimates. Owing largely to the difficulty in sampling and the numerous crops to which GS are pests, action thresholds for individual crops are not well developed. The relationship between GS population density (estimated by sampling methods) and crop health is often influenced by a number of factors, including tillage intensity, crop species, planting date, and crop stage.

In the field, noticeable damage has often been observed if populations exceed an average of five to ten GS per cubic foot (or 1 to 2 GS per 6 x 6 x 12 inch sample) in moderately to highly susceptible crops, such as broccoli, squash, spinach, and cabbage. In conventional cropping systems, pesticides are often applied to susceptible crops if populations exceed three GS per cubic foot. In more tolerant crops, such as potato and small grains, GS feeding may not lead to significant damage, even at considerably higher population densities.

Management and control

For management purposes it is important to make a distinction between tactics that may decrease GS population and those that may reduce crop damage but not necessarily reduce pest populations. In most cases, effective GS management involves establishing a balance between these two tactics. It is important to note that in most cases little can be done without replanting after damage is observed. Sampling is, therefore, important in determining the proper course of action.

Tactics for population reduction

No simple, inexpensive, and completely reliable method of controlling GS has been developed. No method will eradicate GS from a site, and the effect of most tactics will not last longer than one to three years. Very little is known about symphylan population dynamics in agroecosystems due to the complexity of their movements up and down in the soil profile. Many control tactics have been successful in some cases but unsuccessful in very similar situations.

Tillage is probably the oldest control tactic used and is still one of the most effective. Tillage can physically crush GS, thus reducing populations. Tillage may also decrease populations of key GS predators such as centipedes and predaceous mites. However, in annual crops, benefits of increased predator populations in reduced tillage systems have not been shown to be as effective as tillage in decreasing GS populations. In general, for most effective control, till when the GS are in the surface soil, and when soil moisture allows preparation of a fine seed bed. Since only a portion of the population is in the surface horizon, tillage never provides complete control; however, surface populations are commonly significantly lower for at least two to three weeks after tillage. Research suggests that symphylans are more often associated with unbroken down organic matter with good soil structure rather than in compact or sandy soils. There is some evidence of reducing populations and injury by packing down the soil surface after planting; and flooding areas for 2 to 3 weeks.

In conjunction with tillage, pesticides are used to manage GS. Plant protection is probably achieved by direct mortality as well as by repelling GS from the root zone. The use of pesticides has been effective to some degree in conventional systems, but many growers still have perennial problems with symphylans. Pesticides are most effective if applied before planting as broadcast and incorporated applications. Banded/incorporated applications may provide acceptable protection for some crops. In some perennial crops, such as hops, post-plant pesticide applications can reduce GS sufficiently to promote plant vigor. Fumigants, organophosphate, and carbamate pesticides have historically been the most effective, but many are no longer registered for GS in many crops. Pyrethroid pesticides generally do not generally provide as high a level of control. Soil-applied organophosphate insecticides (e.g., Mocap, Lorsban) usually protect crops sufficiently from GS for the production of an annual crop. Soil fumigation, when properly performed, can reduce symphylan populations enough to allow 3 years or more of crop production with no additional control efforts during that period. Refer to individual crop sections for current registrations.

Insecticide registration is continually changing: always check specific insecticide labels for current registered uses. The following may have registered insecticides for symphylan control: asparagus, snap beans, table beets, blueberry, blackberry, broccoli, Brussels sprouts, cabbage, cantaloupe, cauliflower, carrots, celery, chickpea, sweet corn, cucumber, orchard floors, garlic, lettuce, peppers, potatoes, pumpkin, rhubarb, spinach, sugar beets, hops, mint, strawberry, silage and feed corn, clover, grass seed, wheat, barley, radish seed, sugar beet seed, home garden vegetables, home garden strawberries, and home landscape plants.

Crop rotation may partially explain seemingly sudden shifts in GS populations. While GS feed on a wide range of plants, and can even persist in fallow soil, plants vary greatly in their suitability for GS population development. Populations have been shown to decrease significantly in potato crops, even allowing subsequent cultivation in rotation of susceptible crops. Though at this point no other crops have shown to be nearly as effective as potato, numbers have also been found to be lower after a spring oat (‘Monida’) winter cover crop than after a mustard (‘Martiginia’), barley (‘Micah’), or rye (‘Wheeler’) winter cover crop. Mustard and spinach crops have been shown to be very good hosts, and may lead to increasing populations in some cases.

Little information is available on the effect of natural enemies on symphylan populations, which include fungal pathogens, predaceous mites, ground beetles, centipedes, and spiders.

Tactics for crop damage reduction

Most plants can tolerate some level of GS feeding during all or part of the growing season, and numerous tactics can be used to grow healthy crops successfully in GS-infested soil. These tactics can be classified as those aimed at 1) reducing crop damage under high GS populations and 2) reducing the number of GS on crop roots during establishment, when plants are often most susceptible.

Susceptibility to GS feeding can vary dramatically among different soil types, plant species and crop cultivars. Generally, smaller seeded crops tend to be more susceptible than larger seeded crops. Commonly damaged crops include broccoli and other brassica crops, spinach, beets, onions, carrots, corn, and squash. For some crops (e.g. squash), damage can be reduced by increasing the plant density. This can dilute the number of GS per plant and increase survival of young seedlings during highly sensitive stages. The stand can be thinned after establishment, if needed. Beans and potatoes are rarely damaged even under high GS populations. Perennial crops, such as strawberries, raspberries, blueberries, hops, and bare root trees can also be damaged, particularly during establishment. Within a crop, susceptibility is often related to the stage of the crop planted. For example, direct-seeded tomatoes are generally more susceptible than transplants. Broccoli transplants, conversely, often fail to establish under high GS populations.

Garden symphylans are quite active and surprisingly mobile for their size, moving vertically and horizontally through the soil profile. They rely on soil pores and channels made by roots and other soil organisms in order to move through the soil. Therefore, access to roots is strongly correlated with soil structure, bulk density, or “fluffiness” of the soil and pore connectivity. Some tactics focus on temporarily reducing the number of GS in the surface soil, then planting, thus allowing these plants to establish while GS densities are low.

Tillage is an important tactic for decreasing populations in the surface soil. Along with directly killing garden symphylans, tillage breaks apart soil aggregates, modifying soil pores and pore connectivity. The effects of tillage may vary with the type of implements used. In general, the more disruptive the tillage the greater effect it will have on GS movement and feeding. Plowing or disking, followed by thorough preparation of a fine seedbed with a rototiller or roterra, often reduces surface feeding GS populations for two to three weeks. Light rolling, with a landscaping roller or similar implement, is used under some conditions to reduce the size and/or number of macropores, thereby restricting GS movement.

Slug Control

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Slugs are some of the most common and persistent pests of home gardens and commercial crops in western Oregon and Washington, and if left unmanaged can cause significant damage. Slugs are closely related to snails but have no external shell. They are active above ground by day or night, whenever the relative humidity in their immediate environment approaches 100 percent, the temperature rises above 38°F, and the wind speed is negligible. By day, slugs are usually found in the soil, in crevices and cracks, or under soil surface debris where it is moist. Thus, slugs tend to be active primarily at night, but they also feed and reproduce by day during rainy spells, foggy periods, or after irrigation. Even in the summer, when air temperatures peak in the Pacific Northwest and soils seem dry on the surface, slugs can be active at night in closed canopy crops such as legume seed, forage crops, or sugar beets. This is because as night temperatures drop, the humidity of the air between the canopy and the soil often increases, if only for a few hours, even in non-irrigated settings. This "extra time" for feeding and reproduction can eventually lead to very large slug populations. Slugs are relatively inactive when immediate temperatures drop below 38°F or rise above 88°F. They take cover during windy periods and driving rain. Be aware that supplemental irrigation, post-harvest residue buildup on soil surfaces and crop plant structures (e.g., closed canopy) can affect the microclimate of a crop and promote otherwise unexpected slug activity.

Slug damage can be distinguished from that of cutworms and other pests by the presence of slime trails and their small sausage-shaped feces.on the damaged plants as well as on the soil surface around damaged plants. Underground feeding on roots and tubers is characterized by shallow (0.12 inch) to deep (0.5 inch), smooth-sided pits that are usually less than 0.5 inch in diameter. Leaf damage is typified by removal of plant tissue between veins. Seedling grasses and legumes may disappear when slugs feed in the furrow and destroy the growing points of seedlings. In cereal crops, slugs favor newly planted seeds. Wheat is most susceptible to slug damage from seeding to plant emergence.

Slug damage to vegetable and cereal crops, grasses and legumes can be extensive around field margins. Weedy, grassy or wooded borders serve as excellent habitat for slugs. Grass seed crops, cereals, or vegetable plantings that immediately follow a perennial legume or pasture are quite likely to sustain slug damage. Large populations of the gray field slug, and smaller numbers of several less common species build up on most perennial legumes in western Oregon and Washington.

In addition to plant damage, verifying that slugs are present and in damaging numbers in a home garden or a field is usually done by putting out slug bait in late afternoon and returning early the next morning to check for slugs or slime. Put out half a dozen bait stations in the yard or field. Scrape a small area (12 x 6 inches) of the soil surface free of vegetation and debris (making it easier to see small slugs), and scatter four to six pellets of bait inside. You can cover the areas with a scrap of wood or an old carpet tile. This prevents other creatures from disturbing the bait, and the cover helps to keep slugs sickened by the bait from moving away. Place bait stations after the first inch of rain fall in September or early October when slugs become active on the soil surface after having passed the summer underground. Late September to mid-October are usually good months to control slugs, however depending on the weather, other windows of control may occur to apply "follow-up" bait. After October, or when weather becomes too cold (< 34°F) and rainy, baits are less effective, and slugs are not moving or feeding above ground. In these cases bait use is not advised. As days shorten, eggs are produced and these can hatch in fall or when the temperatures warm in the spring. In late fall and early spring, the new juvenile slugs are difficult to spot in the field but can cause significant damage to your crop.

Our most economically important species is the "gray field slug" or gray garden slug (*Deroceras reticulatum)*. The European black or red slug (*Arion rufus),* and in recent years the white-soled slug (*Arion circumscriptus)*, the hedgehog slug (Arion intermedius), the dusky slug (*Arion subfuscus)*, the black greenhouse slug *(Milax gagates)*, the large spotted garden slug (L*imax maximus)*, the marsh slug *(Deroceras laeve)*, and the reticulated slug *(Prophysaon andersoni),* can also be important pests.

Slugs are hermaphrodites: every slug is born with both male and female reproductive parts and theoretically capable of laying eggs. Mating occurs primarily in the fall and spring. Small, round, pearl-like, white or translucent eggs are laid in clusters of a dozen or more (over 500 eggs in a lifetime) in sheltered cavities near the soil surface or under debris on the soil surface if the soil is moist. They typically hatch within 2 weeks to a month. Occasionally, these eggs overwinter if they are laid in late October or November. The greatest egg-laying activity in non-irrigated environments usually occurs after fall rains and again in the spring. The life expectancy for the gray field slug is from 6 to 18 months, but other slug species may live longer.

Chemical control

Slug baits are poisons and therefore can be dangerous to children, pets, wildlife and edible crops. It is important to use baits properly, follow all label instructions and heed all label warnings. Metaldehyde (e.g., Durham®, Deadline M-Ps, Metarex®, Slug Fest®), methiocarb (e.g., Mesurol®), iron phosphate (e.g., Sluggo®, Sluggo Plus®, Natria®, Slug Magic®, Escar-Go® and Worry Free®), and iron chelate (e.g., IronFist®, Ferroxx®, sodium ferric EDTA®) are four common and effective chemicals used to control slugs in the Pacific Northwest. Pellet baits have been the most commonly used product for homeowners. Unfortunately, even when "good" control is achieved, only about 60 to 70 percent of the slug population may be removed. This usually suffices for economic crop protection if slug pressure is light, but does allow the population to recover over time.

Under favorable conditions, slugs can significantly damage a seedling crop in just 1 or 2 days. As the crop emerges (or in the case of cereals, as the seed swells with moisture soon after planting), slugs begin significant feeding. Therefore, application timing, the amount of bait used, bait density (number of pellets per square foot), and bait quality are crucial for successful treatment.

In cereal crops, the greatest risk comes during the first week after planting. Gray field slugs are attracted to the seed furrow and begin to hollow out the endosperm within hours after the seed swells with moisture. One medium-size slug can destroy 10 to 15 wheat seeds before seedlings emerge. Depending on slug density, baits may be applied prior to planting, at planting (broadcast or in the furrow), and shortly afterward. In broadleaf crops and grasses, slugs do not feed on seeds but instead make short order of seedlings by feeding upon and destroying the tender growing points. The most effective timing for bait in these crops is at planting (if slugs are active) or just before seedlings emerge, as this is the most vulnerable plant stage. Preventive treatments are advisable on fields with a long history of slug damage or in no-till situations.

The more effective commercially available baits contain cereal bran or flour as an attractant and are formulated into pellets much smaller than the pencil-eraser-size pellets of the past. These so-called mini pellets, or shorts, are smaller and allow for more pellets per unit area than the larger baits. For instance, some slug bait pellets (e.g., Metarex) are a uniform 2.5 mm long. Look for slug bait in which the pellets are uniform in size, have a high bulk density, are food-based (i.e., smell strongly like cereal to attract slugs from a distance), contain bitrex to prevent unintentional ingestion by mammals, birds and house pets, and are relatively dust free. The result upon broadcasting these pellets is a very dense and uniform pellet distribution per unit area treated. This is significant because slugs tend to encounter these pellets at a greater frequency than the larger, older style type. Research in the PNW indicates that a pellet density approaching 5 to 6 per sq ft is an optimum density. This density can be achieved by applying a per-acre rate of just 5 lb of a 2.5 mm bait, or about 8 to 10 lb of the mini-pellet bait. Doubling or tripling the bait density does not necessarily increase control proportionally. Generally, it is recommended to reapply bait in 10 to 14 days if slug pressure persists, plant damage continues, all bait has been consumed, or the bait has broken down (due to weather). Be sure that the label on the bait product applied will allow for reapplication if needed within this time frame.

The kill rate of a pellet depends on the attractiveness and quality of the carrier, weather conditions at the time of application, and the toxicant level of the bait. If the carrier material is not attractive and palatable to the slugs, they may refuse the bait or consume a sublethal dose of toxicant, from which they can recover.

Methaldehyde, iron-phosphate, iron-chelates, and methiocarb

Several chemicals are formulated into slug and snail baits for use on food and seed crops. Metaldehyde has been used since the early 1940s, iron-phosphate since 1998, and iron-chelates since the early 2000s. Major efforts have been applied to finding new chemical baits for managing slugs in agriculture. The newest generation of products has been developed from metal chelates incorporated into an ingestible bait. These iron chelate baits (IronFist® and Ferroxx®) have been trialed in Oregon and showed positive results in terms of reducing feeding and slug control in grass seed, clover seed, and cereal production.

Baits that contain methiocarb can be effective but they have had limited labels and are used primarily in nonfood or ornamental crops. For example, Mesurol® 75W is used as a spray in nonfood crops and also has activity on certain insect pests as listed on the Gowan label.

Currently a couple of commercial iron-phosphate formulations, Sluggo® and Sluggo Plus®, are approved for organic production. They are formulated as a uniform and dust-free cereal-based mini pellet. Mortality is somewhat slower (5 to 7 days) compared to that induced by metaldehyde. The slugs, however, cease feeding after having eaten the iron phosphate bait. Agricultural use of this product has shown it to be as effective in controlling gray field slugs as metaldehyde baits, although slightly greater rates of the iron-phosphate formulations per unit area are usually needed. Slugs that ingest iron phosphate or iron chelate baits usually die underground or under a source of cover, and not above ground as happens when metaldehyde is consumed.

Metaldehyde is available in various formulations for slug and snail control. These include liquids, sand granules as well as traditional cereal-based baits. Meal formulations (for home use, usually a 2 percent metaldehyde pellet with an insecticide to control other pests) are also available. Liquid metaldehyde and meal formulations may give fast plant protection due to the good coverage, but they do not last more than 2 or 3 days, at best, because wind, UV light and moisture cause metaldehyde to degrade into non-mollusk-killing compounds. Slug Fest® is one such liquid sprayable product and is labeled for use on many food as well as nonfood and ornamental crops. It is often used to control immature slugs prior to canopy closure in establishing a stand.

Big pellets containing metaldehyde need higher application rates for good coverage. They usually provide good control in the first few days, but often degrade quickly and do not persist as long as minipellets. Cereal-based minipellets and very small pellets, (e.g., Metarex) have the best performance record in our rainy climate and can last 2 to 3 weeks on wet soil.

Research has recently shown that metaldehyde has a different mode of action than previously suggested: it does not dehydrate but rather destroys the mucus-producing system unique to slugs (and snails), which severely reduces their mobility and consequently promotes their dehydration through exposure to the sun. Wet conditions, therefore, do not reverse the toxic effect of metaldehyde, as was once thought. However, if slugs do not consume a lethal dose of metaldehyde, they may recover, particularly during wet weather which reduces the likelihood of dehydrating sickened slugs. Furthermore, under wet conditions, poor control may follow from low-quality baits and low active ingredient levels in the bait. This is usually because of rapid (2 to 3 days) physical degradation or fungal growth on pellets that reduces slug feeding.

Due to metaldehyde's specific mode of action, beneficial organisms (earthworms or predatory insects) are not directly affected by baiting with metaldehyde even when these organisms feed on the bait. However, when applying an insecticide such as carbaryl to control certain insect pests like cutworms, armyworms, or wireworms, many beetle predators that feed on slugs, along with earthworms and harvestmen (daddy long-legs), may be killed as well. Be aware, too, that metaldehyde baits are a leading cause of accidental poisoning and deaths of dogs in the PNW.

In western Washington and Oregon, slug control is a year-round necessity in many crops and sites with no-till or conservation tillage practices. Presume damage from slugs in certain crops and sites with a history of problems. Bait early if slug activity is apparent. In some cases, it may be best to bait for slugs before you work the soil (particularly if tillage is shallow and light). Irrigate before baiting in home gardens in order to bring more slugs to the surface during the night. In vegetables, such as Brassicas, baiting must be done before the buttons form or canopy closes, because once the slugs have a chance to enter the head, they are less likely to be attracted to the bait.

Fall baiting usually is recommended for non-irrigated crops. Apply bait after the first rain showers of the season, when slugs become surface active after a summer of hiding deep in the soil to avoid high temperatures and dry conditions. Bait applied immediately after the first fall rains can kill large populations of field slugs before they lay eggs. However, spring applications are also necessary in most fields with minimum or no-tillage practices. Of the eggs that are laid in the fall, some will hatch in 2 to 4 weeks; the rest will hatch during winter or early spring. These newly hatched slugs often do not accept bait as readily as larger slugs.

Control is seldom, if ever, complete. Around the home garden, removing debris, leaf litter, and other excess vegetation helps to remove slug habitat and reduce slug numbers.

Alternative control

Barriers
Various materials, such as salt-impregnated plastic strips and copper strips, provide a small-scale barrier that can work for a few days to a few weeks in keeping slugs away from plants. These barriers have been used with varying degrees of success. For example, underground slug movement or environmental degradation of the repellent (e.g., copper oxidizes, salt washes away) negatively impacts efficacy.

Cultivation
Slug populations can be reduced by tillage. Typically, slug numbers increase when the amount of minimal/zero tillage is increased. Plows, discs, and rototillers crush and bury slugs, disrupt their pathways, expose their eggs to desiccating conditions, dry soil, and remove volunteer-plant food for slugs. Control is proportional to tillage frequency, depth, and efficiency. Plowing followed by disking can be sufficiently effective, so that no further control is needed. A fine seedbed will protect seeds and help prevent slugs accessing seedlings before emergence. Take steps to ensure that a crop has the best chance to emerge from the ground quickly.

Biological control
Many birds, such as starlings, blackbirds, and killdeer, feed on slugs throughout the fall and winter months. Some predatory ground beetles and rove beetles feed on slugs. Naturally occurring fungal and bacterial pathogens, parasitic nematodes, and marsh fly larvae are potential biological control agents of slugs but are not commercially available for use in the United States at this time.

Some nematodes are lethal to slugs and snails, and one species, Phasmarhabditis hermaphrodita, has been used successfully in Europe as a commercially available biological control agent (Nemaslug®). This nematode is associated symbiotically with a bacterium that uses an endotoxin that kills a wide range of pest slugs and snail. After the slug dies, the nematodes multiply over the decaying slug body and then migrate back into the soil to infect more slugs if conditions are favorable. Phasmarhabditis hermaphrodita was recently found in Oregon and California, but Nemaslug® is not available in this country due to biosecurity reasons. Research focusing on discovering and testing pathogenic nematodes in the PNW will likely prove to be valuable for developing biological control agents for slugs and snails.