Small Grains Commodity-based Survey Reference

Cooperative Agricultural Pest Survey (CAPS)
December 2007

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Draft Log

December 2007- Draft sent for CPHST review

February 2008 – Draft sent for CAPS comment

April 2008 – Revised Working Draft posted on NAPIS

May 2008- *Heterodera filipjevi* added to working draft – Revised Working Drafted posted on NAPIS.

Introduction to the Reference

History of Commodity-Based Survey

The Cooperative Agricultural Pest Survey (CAPS) community is made up of a large and varied group of individuals from federal, state, and university organizations who utilize federal and other funding sources to survey for and diagnose exotic and invasive plant pests if possible. By finding pests early, eradication efforts will likely be less expensive and more efficient. For more information on the CAPS and other Plant Protection and Quarantine (PPQ) pest detection programs see: http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/index.shtml.

Traditionally, states have been given a list of pests. Each year, states use this list and choose a number of pests to incorporate in their own specialized surveys. There is certainly value in surveying for plant health threats in terms of discreet pests. However, this approach may not always be the most efficient means of survey. For example, a single pest may occur on a myriad of different hosts, making a comprehensive survey too time consuming and expensive. An alternative method has been suggested. Grouping important pests under the umbrella of a single commodity could be a more efficient way to look for certain pests. The rationale for choosing a commodity survey in certain instances includes the following:

- Survey area will be smaller and targeted.
- Resources can be better utilized with fewer trips to the field.
- Commodities are easy to prioritize in terms of economic and regional (geographic) importance.

The Center for Plant Health Science and Technology (CPHST) has been charged to develop a commodity-based survey strategy in support of the CAPS program. There are two types of end products being developed for each commodity. Each product serves a valuable yet unique purpose. The result is a set of paired documents developed for each commodity. A description of these documents is provided below:

Commodity-based Survey Reference (CSR): This document is composed of a series of pest data sheets, mini-pest risk assessments (PRAs), or early detection PRAs. The data sheets are highly graphic and illustrate the biology, survey, and identification of particular pests in appropriate detail for CAPS surveyors. The pests in this document are numerous. The pests were chosen primarily from the CAPS Analytical Hierarchy Process (AHP) prioritized pest list (CAPS Pest Universe) and the Select Agent list (http://www.cdc.gov/od/sap/docs/salist.pdf or

http://www.aphis.usda.gov/programs/ag_selectagent/ag_bioterr_toxinslist.html). The CAPS pest universe list for FY 08' and 09' are also given in Appendices C and D and is available on the CAPS restricted website for those that have access (http://ceris.purdue.edu/caps/adm2008/ad2008000019.pdf). Additional pests may be added if they are cited in the literature as being a primary pest of the given commodity and are exotic to the United States. Pests may also be added at the request of the CAPS community if deemed to be a primary pest or a regulated pest of small grains. States are not required to survey for all of the pests in this document, but may choose those that are particularly relevant to include in their survey. In general, this document should serve as a desk reference for survey specialists as they plan their cooperative agreements. It may also be useful for obtaining high quality scientific information quickly during the field season.

Commodity-based Survey Guidelines (CSG): This document is smaller. The list of pests is shorter than those chosen for the CSR. A subgroup of the CAPS National Committee determines which pests from the CSR will be included in the CSG. As such, states that participate in these surveys must survey for all organisms listed in the CSG. The CSG set forth guidelines for survey and identification from a broad scale (site selection, number of acres to survey, number of samples to collect, etc.) and a narrow scale (field methods, survey tools, transporting samples, etc.). States are encouraged to follow the procedure set forth in the CSG. The methods are intended to increase the homogeneity of the national data set and increase the statistical confidence in negative data (*e.g.,* demonstration of "free from" status).

As a pilot project, citrus was undertaken as the first commodity in this initiative. The products were developed for implementation in the 2007 survey season. Citrus was chosen, because it is an economically important commodity that is equally distributed in both PPQ regions but is distributed in few overall states. To date, survey strategies for pests of citrus are also well documented. Shortly after completion of the citrus CSG, several other commodity survey guidelines were initiated, including soybean, cotton, grape, and oak forests.

Small Grains Commodity-based Survey Reference

The *Small Grains Commodity-based Survey Reference* (CSR) is a companion document to the *Small Grains Commodity-based Survey Guidelines* (CSG). Both documents are intended to be tools to help survey professionals develop surveys for exotic pests of small grains. The *Small Grains CSR* is a collection of detailed data sheets on exotic pests of small grains. Additionally, the authors have identified native pests that may be easily confused with these exotic pests as well as potential vectors of exotic pests. These data sheets contain detailed information on the biology, host range, survey strategy, and identification of these pests. The commonly confused pests and vectors are included in a section of the pest data sheet dealing with the target pest. Pest specific lures available from the USDA-APHIS-PPQ-CPHST lab in Otis, Massachusettes are also indicated in

the survey section of the document. An order form with current contact information for the OTIS laboratory is given in Appendix F.
By comparison, the *Small Grains CSG* companion document is intended to help states focus resources on survey efforts and identification of a smaller group of target pests (usually less than a dozen). The *Small Grains CSG* contains littleinformation about biology. Instead, they focus on survey design, sampling strategies, and methods of identification. There is no single survey that would be wholly applicable to each location in the United States. Environment, personnel, budgets, and resources vary from state to state. Thus, the *Small Grains CSG* will provide a template that states can use to increase the uniformity and usability of data across political, geographic, and climatic regions while maintaining flexibility for specificity within individual regions.

Purposes of the Small Grains CSR

- To relate scientific information on a group of threatening pests.
- To facilitate collection of pest data at a sub-regional, regional, and national level versus data collection from a single location.
- To aid in the development of yearly surveys.
- To help CAPS cooperators increase their familiarity with exotic pests and commonly confused pests that are currently found in a given commodity.
- To aid in the identification and screening of pests sampled from the field.
- To collate a large amount of applicable information in a single location.

End Users

As previously noted, this document may be used for many purposes. Likewise, it will be of value to numerous end users. As the document was developed, the authors specifically targeted members of the CAPS community who are actively involved in the development and implementation of CAPS surveys.

State Plant Health Director (SPHD): The SPHD is the responsible PPQ official who administers PPQ regulatory and pest detection activities in his or her state. The SPHD is also responsible for ensuring that the expanded role of CAPS is met in his or her state. In many states, the SPHD provides guidance for the State's ongoing management of pest risk and pest detection. However, SPHD responsibilities will vary according to the extent to which each state carries out the various components of the CAPS program.

State Plant Regulatory Official (SPRO): These individuals are employees of their respective states and generally manage the expanded survey program. The SPRO is the responsible state official who administers state agricultural regulatory programs and activities within his or her respective state.

Pest Survey Specialists (PSS): The PSS, a PPQ employee, is supervised by

the SPHD of the state in which he or she is assigned. A PSS may also be responsible for survey activities and may work with the SSC and the surveycommittee in more than one State.

State Survey Coordinators (SSC): The SSC is a state employee responsible forcoordinating each state's CAPS program, participating as a member of the state CAPS committee (SCC), and acting as liaison with the state PPQ office.

Diagnosticians: Diagnostic capabilities vary by state. Some states have advanced networks of diagnosticians, whereas other states access diagnostic support through National Identification Services (NIS) or through contracts with external partners. States are encouraged to utilize qualified diagnosticians in their respective states if expertise is available. PPQ offers diagnostic support for the CAPS program through NIS. A major responsibility for NIS's Domestic Identifiers is to provide diagnostic support to CAPS programs. There are plant pathology and entomology domestic identifiers in each of the regions. A Forest Entomology Domestic Identifier oversees both regions. To learn more about diagnostic resources available to you, discuss your diagnostic requirements and options with your State Plant Health Director, one of the regional Domestic Identifiers, and/or NIS. Appendix A has a listing of NIS and Domestic Identifier contact information.

Organisms Included in the Small Grains Survey Reference

Organisms included in the small grains survey reference are organized first by:

1. Pest type, (*e.g.,* arthropods, plant pathogens, nematodes, and mollusks).

2. Organisms are then divided by their pest status on small grains [*e.g.,* primary pest (major pest) and secondary (minor pest)]. Primary and secondary is determined by reviewing the literature, host association, yield loss, and etc. associated with the pest on a given commodity.
    a. All **primary** and **secondary** pests are CAPS targets, have been through a rigorous prioritization process, and have been determined to pose a threat to the United States. For all primary pests a full, detailed data sheet is included in this manual; while secondary pests have a truncated data sheet. The truncated data sheets focus primarily on symptoms/signs present, survey information, and key diagnostics.
    b. A third group, **tertiary** pests**,** are included with names and photos only to show potential national threats that are not currently CAPS targets, have not been through the rigorous prioritization process, are exotic to the United States, and could be encountered on small grains.

3. Finally, organisms are arranged alphabetically by their scientific names. Common names are provided as well

Previous manuals have included pests from the Eastern and Western Region pest lists. The restructuring of the CAPS program and shift from regional guidelines to a single set of national guidelines has made these lists obsolete. Therefore, pests from these lists were not included in this CSR. States now have more flexibility to survey for pests of state concern, and most regional pests were captured in one or more state CAPS pest lists.

To help provide a rationale for the inclusion of each pest in the reference, the authors have included a section titled, “Reason for Inclusion in Manual”. Pests are either considered to be a CAPS target and are listed in the CAPS prioritized pest list or a national threat. The pests considered as national threats are not known to be present in the United States; however, they are not associated with the CAPS prioritized pest lists but are found on another list. An additional category, requeseted by the CAPS community, is present in some manuals if a pest if suggested that is a primary pest and exotic to the United States or is of regulatory significance.

Introduction to Small Grains

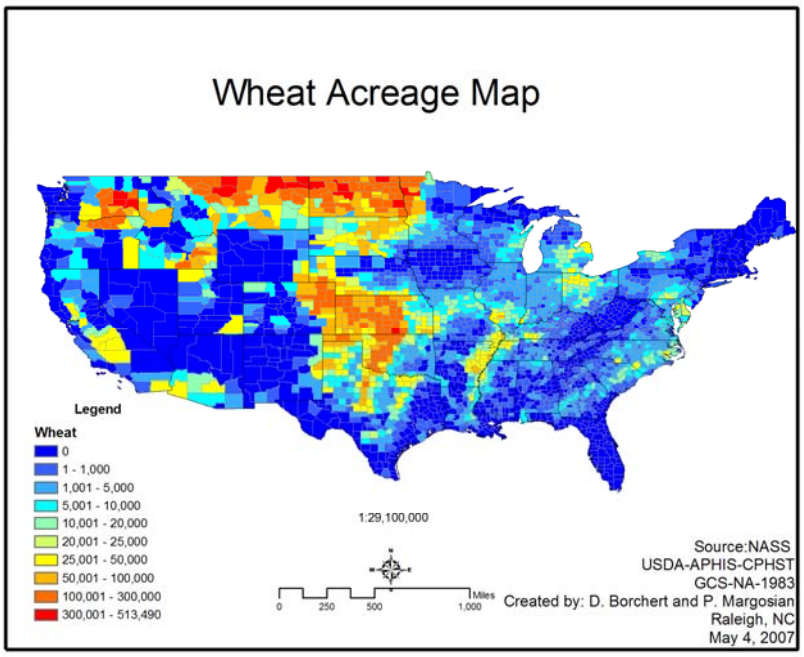
The small grains, wheat, barley, oats and rye, are collectively some of the most important food and feed crops in the United States. All of these important crops are grasses in the family *Poaceae* and are cultivated worldwide.

Wheat (*Triticum aestivum* and *Triticum* spp.) is one of the most important food crops and is used to make flour for a number of foods as well as materials used in fermentation. Various types of wheats are grown for different purposes and are planted in both winter and spring. The total value of the 2006 U.S. wheat crop was over $7 billion. Barley (*Hordeum vulgare*) is an important animal feed and is used in malting and brewing and as a human food. The value of the 2006 U.S. crop was nearly $500 million. Oat (*Avena sativa*) is used both as a human food and as feed for certain types of livestock. The value of the 2006 U.S. oat crop was over $174 million. Rye (*Secale cereale*) is used as a forage crop as well as for food and feed. The U.S. crop was valued at nearly $24 million in 2006. Taken together, the value of production of the U.S. small grains crop is a significant portion of plant production agriculture in the United States and thus, protection against losses from pests that reduce yield, quality, or the ability to export the product is important.

This manual describes 19 pest and pathogen species that have potential or demonstrated ability to reduce crop yield and/or quality for small grains. The four crop species were treated as a group in this manual because of the similar pest problems and agronomic practices. Related crop species like rice will be reviewed in a separate manual.

The manual contains information that is useful in survey, detection and diagnosis of these pests and pathogens. Each section is laid out using a common format. Because of the dynamic nature of pest and pathogen biology, the information contained within this manual should be considered as a starting point and will be updated as new pests are identified, research reveals changes in pest biology, or new diagnostic and survey tools are developed.

Acreage of each of the four small grains by county is shown in the preceding figures.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Legend
Wheat
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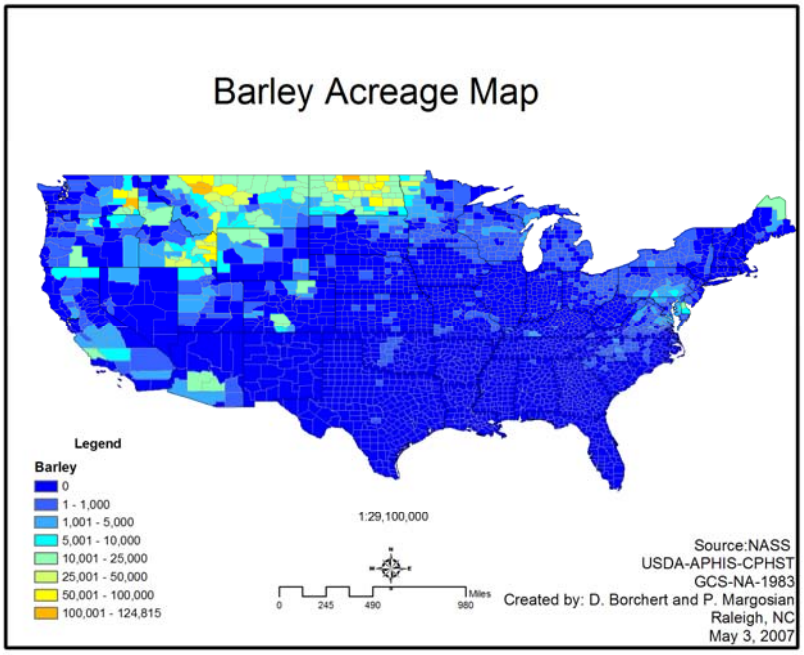
1,001 - 5,000
5,001 - 10,000
10,001 - 20,000
20,001 - 25,000
25,001 - 50,000
50,001 - 100,000
100,001 - 300,000
300,001 - 513,490
1:29,100,000
0 250 500 1,000 Miles
Source:NASS
USDA-APHIS-CPHST
GCS-NA-1983
Created by: D. Borchert and P. Margosian
Raleigh, NC
May 4, 2007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Legend
Barley
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25,001 - 50,000
50,001 - 100,000
100,001 - 124,815
1:29,100,000
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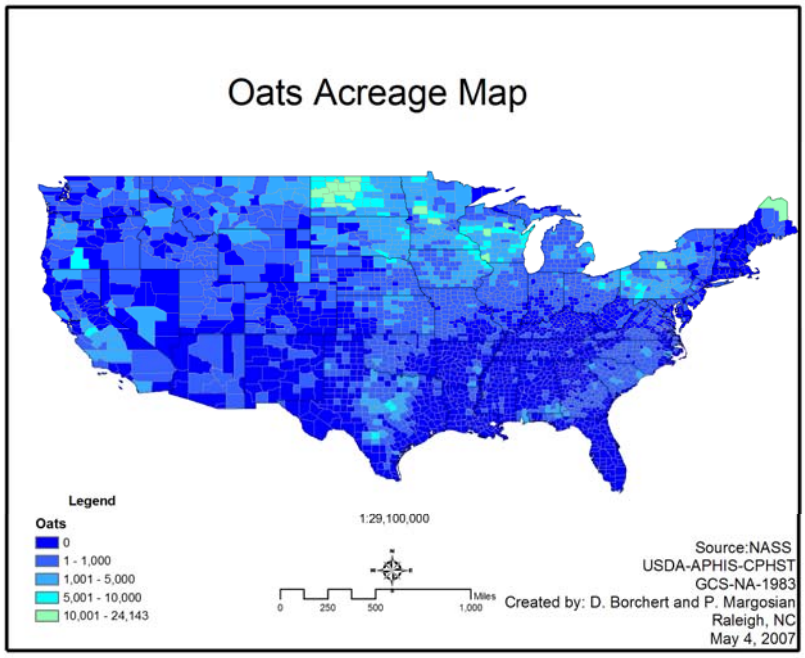
USDA-APHIS-CPHST
GCS-NA-1983
Created by: D. Borchert and P. Margosian
Raleigh, NC
May 3, 2007


Oats Acreage Map
Legend
Oats
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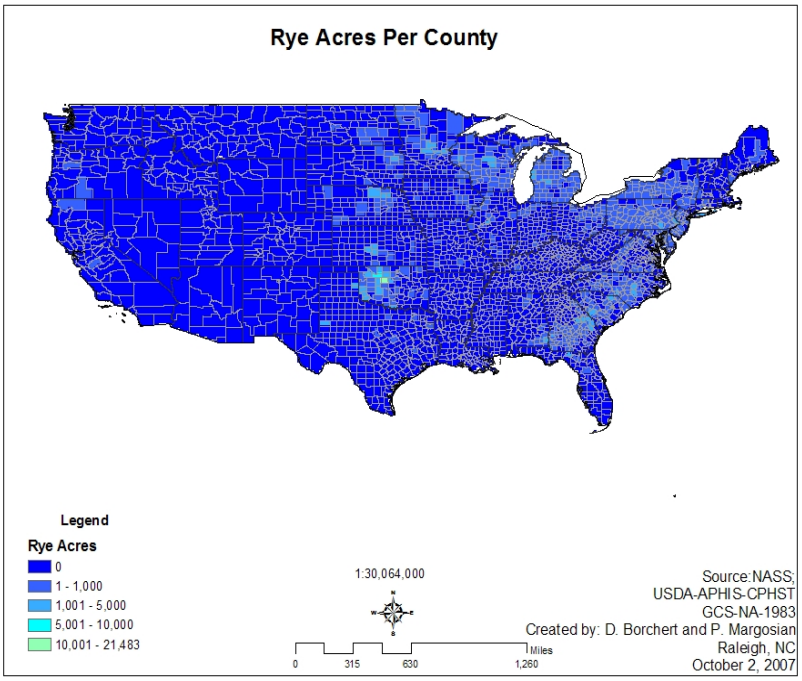
1 - 1,000
1,001 - 5,000
5,001 - 10,000
10,001 - 24,143
1:29,100,000
0 250 500 1,000
Miles
Source:NASS
USDA-APHIS-CPHST
GCS-NA-1983
Created by: D. Borchert and P. Margosian
Raleigh, NC
May 4, 2007


Rye Acres Per County
Legend
Rye Acres
0
1 - 1,000
1,001 - 5,000
5,001 - 10,000
10,001 - 21,483
1:30,064,000
0 315 630 1,260
Miles
Source:NASS;
USDA-APHIS-CPHST
GCS-NA-1983
Created by: D. Borchert and P. Margosian
Raleigh, NC
October 2, 2007

References:

National Agricultural Statistics Service. www.nass.usda.gov

Wiese, M.V. (ed.). 1987. *Compendium of Wheat Diseases*, 2nd edition. APS Press, St. Paul, MN.

Mathre, D.E. (ed.). 1997. *Compendium of Barley Disease*, 2nd edition. APS Press, St. Paul, MN.

Smith, C.W. 1995. Crop Production: Evolution, History, and Technology, John Wiley and Sons, NY. 496 pp.

Arthropods

Primary Pests of Small Grains (Full Pest Datasheet)

Autographa gamma

Scientific Name
Autographa gamma L.

Synonyms:
Phytometra gamma and *Plusia gamma*

Common Name(s)
Silver-Y moth, beet worm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Figure 1. Eggs of *A. gamma*. Photo courtesy of Jurgen Rodeland. http://www.rodeland.de/fotos/lepidoptera/autographa_gamma.htm

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description
Eggs: Semi-spherical, 0.57 mm in diameter. Eggs are yellowish-white (Fig. 1), later turning yellowish-orange to brown. The number of ribs varies from 28 to 29 (Paulian et al., 1975). The eggs are deposited in bunches or singly on the underside of leaves.

Figure 2. Larva of *A. gamma*. Photo courtesy of P. Mazzei. www.invasive.org

Larvae: The larva is a semi-looper with three pairs of prolegs. It occurs in varying shades of green (Fig. 2), with a dark-green dorsal line and a paler line of whitish-green on each side. The spiracular line is yellowish, edged above with green. There are several white transverse lines between the yellow spiracular line and the dorsal black line. Some larval forms have a number of white spots. The head may have a dark patch below the ocelli or be entirely black. Maximum length 20 to 40 mm (USDA, 1958; Jones and Jones, 1984; Emmett, 1980; Hill, 1987).

Pupae: Pupation takes place within a translucent, whitish cocoon spun amongst plant foliage (Fig. 3). The leaves may sometimes be folded over. The pupa is brown to black, greenish or even whitish-green on its ventral side, 16 to 21 mm long, 4.5 to 6.0 mm broad. Cremaster globular, with four pairs of hooklets (Paulian et al., 1975; Carter and Hargreaves, 1986).

Figure 3. Cocoon of *A. gamma*. Photo courtesy of Alain Fraval. http://www.inra.fr/hyppz/RAVAGEUR/6autgam.htm

Adults: The adults are gray-colored and the forewings are marbled in appearance; their color being silvery-gray to reddish-gray to black with a velvety sheen. Wing expanse is 36 to 40 mm. The 'Y' mark on the forewing is distinct and silvery (Fig. 4). The hindwings are brownish with a darker border (USDA, 1958; Jones and Jones, 1984; Hill, 1987).

Figure 4. Adult showing the silver *Y* mark that resembles the Greek letter gamma. Photo courtesy of Jeremy Lee.

Biology and Ecology

Adult moths feed on nectar primarily in the early morning or evening hours, young larvae skeletonize the leaves, and older late-instar larvae eat the entire leaf. The polyphagous larvae damage many agricultural plants and eat the foliage of over 200 plant species (Steudel, 1963).

Females lay from 500 to more than 1000 whitish eggs (Hill, 1987), singly or in small batches, on the underside of leaves of low-growing plants. *Autographa gamma* passes through five larval instars (Harakly, 1975). Depending on the climate of the region, *A. gamma* can have two to four generations per year (Rashid et al., 1971; Dochkova, 1972; Harakly, 1975; CABI, 2004). *Autographa gamma* does not undergo true diapause (Tyshchenko and Gasanov, 1983), but pupae or late-instar larvae can overwinter in moderate climates (Dochkova, 1972; Tarabrina, 1970; Kaneko, 1993; Saito,

1988). In areas where *A. gamma* is unable to overwinter, severe infestations occur sporadically. The longevity of females can be almost twice that of males (Harakly, 1975).

High temperatures decrease the life span (Harakly, 1975), and depending on temperature, *A. gamma* has a life span from 28 to 65 days (Rashid et al., 1971; Harakly, 1975). The incubation period lasts for 3 days at 25°C (Ugur, 1995), but in temperate regions, egg incubation may take 10 to 12 days (Hill, 1987). Larval development takes from 51 days at 13°C to 15 to 16 days at 25°C and the pupal stage from 32 days at 13°C to 6 to 8 days at 25°C (Hill and Gatehouse, 1992; Ugur, 1995).

The lower threshold temperatures for egg, larvae, pupae and pre-oviposition periods range between 9.3-11.0 °C and depends upon plant species used for feed. Giving an average threshold temperature of 10 °C, the degree days varied between 177 to 257 for different plant species (Honěk et al., 2002).

A. gamma is primarily nocturnal. An average-sized moth that is unaided by wind can fly 16km/h for 50 km without feeding; some larger moths can fly over 100 km (Macaulay, 1974). *A. gamma* is migratory and can disperse over distances spanning hundreds of kilometers (Macaulay, 1972, 1974; Harakly, 1975).

Symptoms/Signs

There are no specific symptoms and signs listed in the literature for small grains. Eggs (singly or in small clusters) may be visible on leaves of low growing plants. Larvae are active at night. During the day, they remain pressed against the underside of the leaf; when disturbed they tend to drop off the plant. Leaves may be skeletonized by larval feeding. Older leaves are preferred by larvae. The larvae only eat young leaves after destroying the old ones. The petioles or leaf stalks may be cut by the larvae. Frass may or may not be visible. Pupae are found in the folds of the lower leaves of the host plant. Webbing may be present. Adult moths feed on flowers and can often be seen feeding during the day or early evening (USDA, 1985).

Pest Importance

Outbreaks of *A. gamma* occur periodically over wide areas of Europe, Asia and North Africa. The outbreak of 1928, which occurred in most of central Europe, caused widespread defoliation of peas in Poland. Damage from this insect and *Pieris rapae* (cabbage white) in areas of the Netherlands was valued at as much as 320,000 guilders (~$180,000) during some years in the 1800s. It is also very destructive in England and Denmark (CABI, 2004).

Damage to globe artichokes was severe near Bari, Italy in 1982 to 1985, with about 55% of plants being damaged, and *A. gamma* was one of the major pests (Ippolito and Parenzan, 1985).

Studies in Czechoslovakia (Novak, 1975) indicated that damage became of economic significance when 25% of the leaf area of a plant was destroyed.

Known Hosts

This polyphagous pest is found on cereals, grasses, fiber crops, *Brassica* spp. and other vegetables including legumes. *Autographa gamma* can feed on at least 224 plant species, including 100 weeds, from 51 families (Maceljski and Balarin, 1972). Wheat is considered a primary host.

Major hosts

Beta vulgaris (beet), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Borago officinalis* (common borage), *Brassica oleracea* var. *capitata* (cabbage), *Brassica oleracea* var. *gemmifera* (Brussels sprouts), *Brassica rapa* subsp. *chinensis* (Chinese cabbage), *Brassica rapa* subsp. *pekinensis* (Pe-tsai), *Cannabis sativa* (marijuana, hemp), *Capsicum* (peppers), *Chrysanthemum indicum* (chrysanthemum), *Cicer arietinum* (chickpea), *Cichorium intybus* (chicory), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annuus* (common sunflower), *Hyssopus officinalis* (hyssop), *Lactuca sativa* (lettuce), *Linum usitatissimum* (flax), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Pelargonium* (geranium) hybrids, *Petroselinum crispum* (parsley), *Solanum tuberosum* (potato), *Spinacia oleracea* (spinach), *Trifolium pratense* (red clover), *Triticum aestivum* (wheat), *Vitis vinifera* (grape), *Zea mays* (maize), and *Zinnia elegans* (zinnia)

Known Vectors (or associated organisms)

Autographa gamma is not a known vector and does not have any associated organisms.

Known Distribution

Autographa gamma is widely distributed throughout all of Europe and eastward through Asia to India and China; it also occurs in North Africa (USDA, 1958).

Asia: Azerbaijan, China, India, Iran, Iraq, Israel, Japan, Kazakhstan, Korea, Saudi Arabia, Syria, Turkey, and Uzbekistan. **Europe:** Austria, Belgium, Bulgaria, Former Czechoslovakia, Denmark, Finland, Former USSR, France, Germany, Greece, Hungary, Iceland, Italy, Latvia, Lithuania, Moldova, Netherlands, Poland, Portugal, Romania, Russian Federation, Serbia and Montenegro, Slovakia, Spain, Sweden, Switzerland, Ukraine, and United Kingdom. **Africa:** Algeria, Egypt, Libya, and Morocco.

Potential Distribution within the United States

The likelihood and consequences of establishment by *A. gamma* have been evaluated in a pathway-initiated risk assessment. *Autographa gamma* was considered highly likely of becoming established in the United States if introduced. The consequences of its establishment for U.S. agricultural and natural ecosystems were also rated high (i.e., severe) (Lightfield, 1997).

Survey

Due to the migratory nature of this species, adult *A. gamma* can be observed every month from April to November, usually peaking in late summer (CABI, 2004).

Taken from Venette et al. (2003).
Preferred Method: The sex pheromone, (*Z*)-7-dodecenyl acetate and (*Z*)-7-dodecenol in ratios from100:1 to 95:5, has been used to attract and monitor male flight of *A. gamma*. In field applications, the pheromone may be dispensed from rubber septa at a loading rate of 1 mg (CAPS, 1996). Lures should be replaced every 30 days (CAPS, 1996). Newly-emerged adult males of *A. gamma* are not attracted to the pheromone; 3-day old males are most responsive to the lure. The pheromone of *A. gamma* may also attract other Lepidoptera in the U.S. such as *Anagrapha ampla*, *Anagrapha falcifera*, *Autographa ampla*, *Autographa biloba*, *Autographa californica*, *Caenurgia* spp., *Epismus argutanus*, *Geina periscelidactyla*, *Helvibotys helvialis*, *Lacinipolia lutura*, *Lacinipolia renigera*, *Ostrinia nubilalis*, *Pieris rapae*, *Polia* spp., *Pseudoplusia includens*, *Rachiplusia ou*, *Spodoptera ornithogalli*, *Syngrapha falcifera,* and *Trichoplusia ni.* Trapping is suggested in major truck farming areas. Traps should be placed within or on the edge of fields of the host crops. Traps should be suspended from stakes and placed at the level of crop height and raised as the crop matures. **This lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory in the 100:1 ratio.**

Alternative Method: The USDA (1986) provides some considerations for visual inspections of host plants for the presence of eggs, larvae, or pupae. In general, eggs may be found on the lower and upper surfaces of leaves. Larvae are likely to be found, if left undisturbed, on leaves that have been skeletonized or that have holes in the interior. Pupae may be found on the lower leaf surface (USDA, 1986).

Not Recommended: Adult males and females have also been collected using Robinson black-light traps, but these traps attract moths non-discriminately. Such traps, placed 3 meters above the ground, have been used to successfully monitor the dynamics of *A. gamma* and other Noctuid moths. Sticky traps have been used, but are not recommended as pheromone traps are much more specific and effective.

Key Diagnostics

Species are most reliably identified by close examination of the genitalia (Nazmi et al., 1980; USDA, 1986).

Easily Confused Pests

Several life stages of Noctuid pests can be confused with *A. gamma,* of these, the most important species include: *Trichoplusia ni* (cabbage looper*,* Fig. 5*) Syngrapha celsa (*Fig. 6)*, A. pseudogamma* (Fig. 7*)*, *and A. californica* (alfalfa looper, Fig. 8) that are already present in the continental United States.

The other easily confused species are *Cornutiplusia circumflexa* (Essex Y), which is distributed in Europe, Asia and Africa, and *Syngrapha interrogationis* (Scarce Silver Y) ,which is established in the United Kingdom(Venette et al., 2003). Adults of *A. gamma* are gray to grayish brown in color with a "Y mark or gamma [γ] on the forewing". Nazmi et al. (1981) compare similarities and differences between closely related species.

Figure 5. Adult and larva of *Trichoplusia ni*. Photos courtesy of Keith Naylor and Extension Entomology, Texas A&M University.

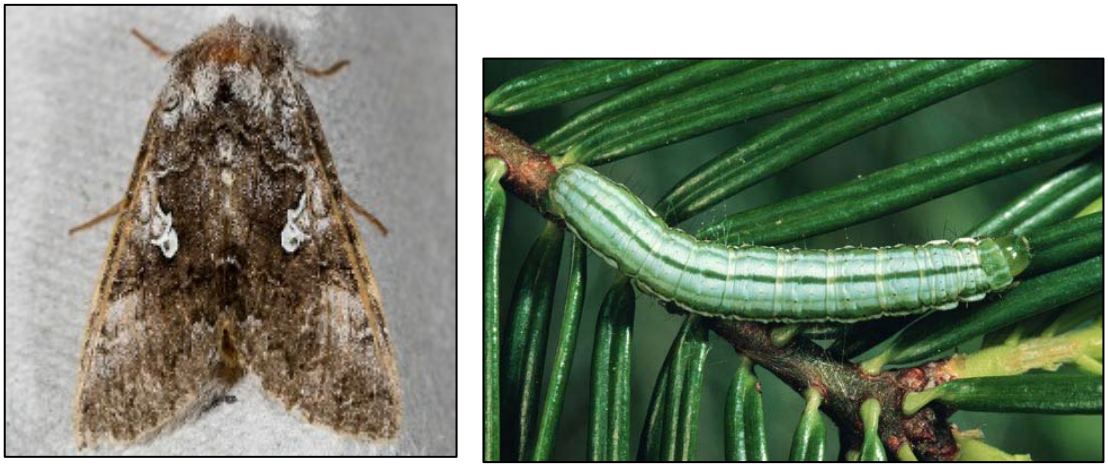


Figure 6 Adult and larva of *Syngrapha celsa*. Photos courtesy of John Cooper and Natural Resources Canada.

Figure 7. Adult of *Autographa pseudogamma.* Photo courtesy of Natural Resources Canada.

Figure 8. Adult and larva of *Autographa californicum.* Photos courtesy Franklin Dlott, UC Cooperative Extension, Monterey County.

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Copitarsia spp.

Scientific Name
Copitarsia incommoda Walker
Copitarsia decolora Herrich-Schaffer

Note: Systematics and nomenclature within the genus *Copitarsia* are particularly problematic. Over time, the genus has included from six to twenty one species, depending on which taxonomic authority is consulted (Angulo and Olivares, 2003; Venette and Gould, 2006). Currently described species include: *C. anguloi*, *C. basilinea*, *C. clavata*, *C. editae*, *C. humilis*, *C. incommoda* (=*C. consueta*), *C. naenoides*, *C. paraturbata*, *C. patagonica*, *C. purilinea*, and *C. decolora* (=*C. turbata*). The validity of the eleven names has come into question. Because *Copitarsia* spp. have not been examined with modern phylogenetic techniques, these names may represent geographic variants of one or two species (Venette and Gould, 2006).

Most of this pest data sheet is at the genus level due to the taxonomic confusion, however, detailed pest descriptions are given for the two most economically important pest species, *C. incommoda* and *C. decolora*

Synonyms:
Copitarsia decolora: *Agrotis heydenreichii, A. hostilis, Copitarsia hostilis, C. inducta, C. margaritella, C. sobria, C. subsignata, C. turbata, Graphiphora sobria, Lycophotia margaritella, Mamestra decolora, M. inducta, Polia turbata,* and *Spaelotis subsignata*.

Copitarsia incommoda: *Agrotis consueta, A. incommoda, A. peruviana, Allorhodecia hampsoni, Copitarsia consueta,* and *C. peruviana.*

Common Names
Owlet moths, cutworms, army worms, leaf worm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description

General (all species): *Copitarsia* eggs are deposited singly or in egg masses (Fig. 1A). A single female may produce between 570 and 1640 eggs, depending on the quality of the environment and the host. Larvae complete five to six instars during development and reach a length of approximately 2 to 4 cm. The larvae tend to be green in color (Fig. 1B), but green, black, and gray phases occur that vary with habitat and crops attacked. Development time from egg to adult depends on many factors including temperature, humidity, and host. Reported larval development times vary from approximately 43 days at 24.5°C on lettuce to 82.5 days at 20.4°C on artificial diet (Arce de Hamity and Neder de Roman, 1992; Lopez-Avila, 1996; Velasquez, 1998).

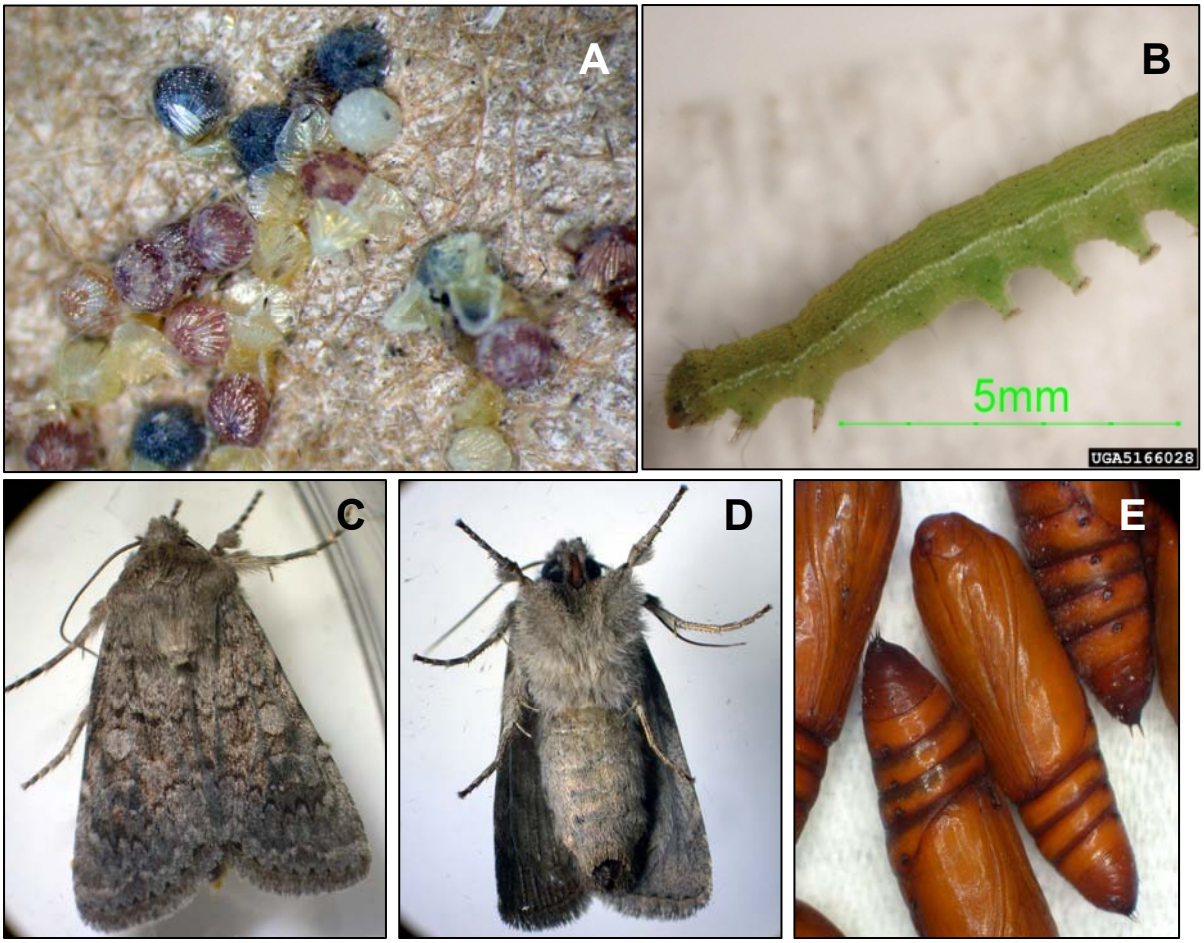

Figure 1. *Copitarsia* life stages. Eggs (A), Larva (B), Adult (C, D), and Pupae (E). Photos courtesy of Rebecca Simmons and Charles Olsen, University of North Dakota (www.und.edu/misc/compitarsia) and USDA APHIS PPQ. (www.forestryimages.org), respectively.

Copitarsia pupate in the soil (Fig. 1E) and emerge as gray or brown moths (Fig. 1C, D) that are difficult to distinguish from other noctuids. Diapause has not been reported for any member of the genus. The literature suggests that *Copitarsia* are

multivoltine through much of the range. In general, *Copitarsia* have two to four generations per year.

Copitarsia decolora: (**From Simmons and Pogue, 2004**)
Description. Medium-sized, light brown or gray moths with well-defined orbicular and reniform spots.

Discussion. *Copitarsia decolora* varies slightly in coloration from lighter to medium brown. Females tend to be larger and have darker hindwings than males. Mitochondrial DNA evidence indicates at least two morphologically cryptic species within *C. decolora:* one ranging from southern Mexico to Ecuador, the other occurring in Ecuador, Colombia and Peru (Simmons and Scheffer, 2004).

Diagnosis. *Copitarsia. decolora* lacks the brush-like androconia found in male *C. incommoda*. Male *C. decolora* have a blunt digitus and corona of spines on the valve. Female *C. decolora* are recognizable due to the speculate, heavily sclerotized antevaginal plate.

Male.

Head. Brown; antenna light brown, biserrate and ciliated; palpus light brown, apex white.

Thorax. Patagium brownish gray; mesothorax pale brown; metathorax gray to white; fore, mid, and hindleg mixed with white and brown scales, tibial spurs striped with brown; tarsi white.

Wings.

Forewing. Length = 13 to 18 mm (average = 16.1 mm, SD = 1.3 mm, *n*= 14). Ground color light brown or gray; antemedial and postmedial lines, double row of brown zigzag lines, with white between them; basal area with well defined brown lines; reniform spot brown outlined in white; orbicular spot ground color with white inner and black outer margin; outer margin with triangular black spots between wing veins; fringe grayish brown.

Hindwing. Ground color white; wide marginal band brown; veins toward wing margin brown; fringe brown basally, remainder white.

Abdomen. First three abdominal segments light gray, remainder of abdomen gray; genital tuft gray; sclerotized patches present in pleural membrane near second abdominal segment; hair brushes, scent pouches and modified S2 absent; terminal tergite weakly sclerotized medially, more heavily sclerotized laterally, forming two circular areas.

Genitalia. Tegumen rounded; uncus apically swollen, bearing long setae; saccus extended into narrow point; valve sinuate, tapering to pointed

apex; corona present; ampulla attenuate, apex extending beyond costal margin of valve; digitus spatulate; juxta a broad plate with pointed lateral margins, medio-ventral plate with rounded, sinnuate margins, dorsal margin V-shaped with a pair of ventrally produced arms with dorsally curved apices; spinose pad present above aedeagus; apex of aedeagus with a small sclerotized plate (sp) consisting of one large and two pointed projections, a large serrate sclerotized plate (lp) opposite small plate; vesica elongate; cornuti various sized elongate spines in both clusters and solitary in a spiral line in basal one-quarter of vesica.

Female. As in male, except antennae filiform and cilated; forewing length = 14 to 18 mm (average = 16.8 mm, SD = 1.2 mm, *n* =24); hindwing darker than males.

Genitalia. Papillae anales, posterior apophyses unmodified; anterior apophyses reduced in length, thickened; S8 unmodified; antevaginal plate U-shaped, spiculate texture, symmetrical; ductus bursae sclerotized, spinose; corpus bursae deeply ridged, spherical, three lines of signa; appendix bursae larger than corpus bursae, membranous, irregular in shape; ductus seminalis from posterior of appendix bursae.

Copitarsia incommoda: (from Simmons and Pogue 2004)

Description. Medium-sized, pale brown moths, with well-defined orbicular and reniform spots, and light brown hindwings.

Discussion. *Copitarsia incommoda* varies slightly in coloration from lighter to medium brown. Females tend to be larger and have darker hindwings than males.

Diagnosis. *Copitarsia incommoda* is often confused with *C. decolora.* Males of *C. incommoda* can be identified externally by their brush-like androconia on the second abdominal segment (sometimes only after dissection), which are absent in *C. decolora.* Male *C. incommoda* has a rounded digitus, and valves lack a corona of spines that are present in *C. decolora.* Female *C. incommoda* can be identified by the smooth texture of the U-shaped antevaginal plate, compared with the spiculate antevaginal plate found in *C. decolora*.

Male.

Head. Brown; antenna pale brown, filiform and ciliated; palpus brown.

Thorax. Patagium brown; mesothorax lighter, tawny brown; metathorax cream to white; fore, mid, and hindleg mixed white and brown, tibial spurs striped with brown, tarsi white.

Wings.

Forewing. Length = 14 to 18 mm (average = 16 mm, SD = 1.3 mm, *n* = 15). Ground color light brown; antemedial and postmedial lines, a double row of brown zigzag lines with white between them; basal area with well-defined brown lines; reniform spot ground color with white inner and black outer margin; orbicular spot ground color outlined in black; outer margin with triangular black spots between wing veins; fringe brown.

Hindwing. Ground color brown mixed with white scales basally; fringe light brown basally, rest white.

Abdomen. Brown, genital tuft white; hair brushes, scent pouches and modified S2 present (Fig. 2B); terminal tergite as in *C. decolora*.

Genitalia. As in *C. decolora,* except corona absent; digitus slender, apex round, not spatulate; apex of aedeagus with a small sclerotized plate (sp) consisting of one large, one small, and three minute pointed projections; a series of variously sized, heavily sclerotized spines opposite small plate (ss); cornuti in a similar pattern to that of *C. decolora,* but more robust.

Female. As in male, except forewing length = 14 to 19 mm (average = 17.2 mm, SD = 1.3 mm, *n* = 18); hindwing darker than males.

Genitalia. As in *C. decolora,* except lateral lobes of U-shaped antevaginal plate larger than *C. decolora*.

Biology and Ecology

Gould et al. (2005) examined the effect of temperature on survival, development, and reproduction of *C. decolora*. *C. decolora* eggs required 66 degree days (DD) to complete development with a base temperature of 7.8°C (46° F). *C. decolora* developed through four to six instars depending on temperature and food source. Development of larvae from neonate through prepupa required 341.4 DD above a base of 7.3° C (45° F) on asparagus, whereas 254.5 DD were needed on artificial diet at the base temperature of 7.7°C (46° F). Pupae required approximately 236 DD (at 8.2-8.4°C (~47°F)) to develop when reared on asparagus or artificial diet.

Female moths laid significantly more eggs at 14.6 and 20.1°C (58 and 68°F, respectively) than at higher and lower temperatures. Survival of individuals to the adult stage increased from 71% at 9.7°C (49 F) to 93% at 24.9°C (77°F). Survival fell off rapidly to 25% at 29.5°C (85°F). The generation time was the shortest at 29.5° C; however, only 25% of females survived to the adult stage, fecundity was low, and only 53% of the eggs hatched. The capacity for increase was low at 9.7°C, peaked at 25.7°C (78°F), and declined as temperatures increased, The authors estimated that the populations on asparagus would not develop at temperatures >31.3°C (88°F) or <6.9°C (44°F).

Symptoms/Signs

Eggs and larvae may be present on plant parts. Larvae generally feed externally on leaves, stems, and fruits of host plants but will occasionally bore into thicker non-woody tissues (Venette and Gould, 2006).

Pest Importance

In South America, *Copitarsia* reduces the marketability of some vegetables by 24% and reduces grain yield by 80 to 90% (Venette and Gould, 2006). *Copitarsia* eggs and/or larvae are often detected at U.S. ports of entry on cut flowers and vegetable commodities. If *Copitarsia* spp. are found in a shipment, the commodity must be treated, destroyed, or returned to its country of origin because it is considered a quarantine pest. *Copitarsia* species are difficult to identify, and border regions have been extensively sampled for the presence of these species.

Two species, *Copitarsia incommoda* and *C. decolora*, are the most economically important members of the genus. *C. incommoda* is reported from Mexico to northern Chile. Documented hosts of *C. incommoda* include asparagus, rapeseed, and alfalfa. *C. decolora* is widely distributed in Central America and South America and has been reported from Mexico to Chile and east to Argentina. *Copitarsia decolora* feeds on a variety of crops, including artichokes, cut flowers, lettuce, peas, beets, cabbage, carrots, corn, beans, and potatoes. *C. decolora* is routinely intercepted on produce at US ports of entry. This species has historically been misidentified as *C. incommoda* in both agricultural and taxonomic literature.

Known Hosts

Polyphagy is common among members of the genus. Thirty nine crop plants are listed as hosts in the published literature, and the genus has been found at U.S. ports of entry on several additional plant species not reported in the literature. Collectively these plants represent 19 families. Because this pest data sheet covers multiple *Copitarsia* spp., determining major and minor hosts is quite difficult; therefore hosts reported in the literature and identified at U.S. ports of entry are simply listed.

Hosts reported in literature: *Actinidia chinensis* (kiwi), *Pistacia* spp. (pistachio), *Coriandrum sativum* (coriander), *Daucus carota* subsp. *sativus* (carrot), *Calendula* spp. (calendula), *Cynara scolymus* (artichoke), *Helianthus annuus* (common sunflower),, *Lactuca* spp. (lettuce), *Ullucus tuberosus* (ulluco), *Brassica napus* (canola), *Brassica oleracea* (cabbage, cauliflower, broccoli), *Simmondsia californica* (jojoba), *Dianthus caryophyllus* (carnation), *Beta vulgaris* (beet), *Beta vulgaris* ssp. *cicla* (chard), *Chenopodium quinoa* (quinoa), *Spinacia oleracea* (spinach), *Vicia faba* (broad or lima bean), *Cicer arientinum* (chick pea), *Medicago sativa* (alfalfa), *Pisum* spp. (peas), *Trifolium pratense* (red clover),

Gladiolus spp. (gladiolus), *Lolium multiflorum* (ryegrass), *Rosmarinus officalis* (rosemary), *Allium cepa* (onion), *Asparagus officinalis* (asparagus), *Linum usitatissimum* (flax), *Triticum aestivum* (wheat), *Zea mays* (corn), *Polygonum segetum* (field smartweed), *Fragaria chiloensis* (strawberry), *Malus* spp. (apple), *Rubus idaeus* (raspberry), *Capsicum* spp. (pepper), *Lycopersicon esculentum* (tomato), *Nicotiana tabacum* (tobacco), *Physalis pubescens* (downy groundcherry), *Solanum melongena* (eggplant), and *Solanum tuberosum* (potato).

Additional plant species identified at ports of entry: *Limonium* spp. (sea lavender), *Alostroemeria* spp. (lily of the Incas), *Dianthus* spp. (pinks), *Chrysanthemum* spp. (chrysanthemum), *Gypsophila* spp. (babysbreath), *Aster* spp. (aster), and *Rosa* spp. (rose). Note: It is not known if the *Copitarsia* spp. were actively feeding or if they were simply hitchhiking on these additional plant species (Venette and Gould, 2006).

Known vectors (or associated organisms)

Copitarsia spp. are not known to be vectors and do not have any associated organisms.

Known Distribution

Copitarsia spp. can be found along the western edge of South and Central America from the tip of Argentina through central Mexico. *Copitarsia* spp. have been reported in the literature from all countries south of the United States except Belize, Brazil, El Salvador, French Guiana, Honduras, Nicaragua, Panama, Paraguay, Suriname, and the islands of the Caribbean. Nevertheless, the genus has been intercepted by USDA, APHIS on commodities shipped from several countries known to have *Copitarsia* spp. and from Belize, Brazil, Dominican Republic, El Salvador, Haiti, Honduras, Jamaica, Nicaragua, Panama, St. Lucia and Trinidad and Tobago (Venette and Gould, 2006). The true origin of these commodities is not known. However, such information suggests that the range of *Copitarsia* extends from central Mexico to southern South America and may include several Caribbean nations.

Potential Distribution within the United States

Populations of the genus have not been reported in the United States. Venette and Gould (2006) estimate that *Copitarsia spp.* may have the potential to become established in 70% of the contiguous United States and are unlikely to be constrained by host availability due to its broad host range.

Survey

Preferred Method: A pheromone consisting of (Z)-9-tetradecenyl acetate (Z9-14:Ac) and Z-9-tetradecenol (Z9-14:0H) has been previously identified for *C. decolora* (Rojas et al., 2006). Captures in traps baited with a mixture of Z9-14:Ac and Z9-14:0H at 4:1, 10:1, and 100:1 ratios were not significantly different from

traps baited with virgin females. The commercial availability of this pheromone, however, is unknown at this time. The same components were identified for *C. incommoda* (Cibrian-Tovar et al., 2003).

Early detection surveys have traditionally utilized non-selective black light trapping.

Alternative Method: Survey for *Copitarsia* spp. generally has been conducted visually at the ports by examining cut flowers and vegetable products destined for entry into the United States. All products are examined for the presence of egg masses and/or larvae.

Key Diagnostics

Adult *Copitarsia* spp. have few external characteristics to distinguish them from other noctuid moths and can only be identified with confidence by genitalia dissections. *Copitarsia* larvae can be distinguished from other genera based on external characteristics. For example, *Copitarsia* larvae have dark bars at the base of the two medial setae, white dorsal setae, misaligned head setae (dorsal ventrally), and two dark triangles on the posterior abdominal segments (Riley, 1998).

Within the *Copitarsia* genus, adults can be identified by the presence of large spines on the foretarsi; however, larval and egg identification characters are inconsistent or nonexistent (Simmons and Pogue, 2004). Angulo and Olivares (2005), however, state that *C. incommoda* and *C. decolora* larvae can be distinguished by examining the spinneret and pinnaculae.

Easily Confused Pests

Adult *Copitarsia* spp. have few external characteristics to distinguish them from other noctuid moths and can only be identified with confidence by genitalia dissections. At times, members of *Copitarsia* have been confused with the genera *Agrotis*, *Euxoa*, *Polia*, and *Orthosia* (Venette and Gould, 2006).

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Helicoverpa armigera

Scientific Name
Helicoverpa armigera Hübner

Synonyms:
Bombyx obsoleta, Chloridea armigera, Chloridea obsolete, Helicoverpa commoni, Helicoverpa obsolete, Heliothis armigera, *Heliothis conferta, Heliothis fusca, Heliothis obsoleta*, *Heliothis pulverosa*, *Heliothis rama*, *Heliothis uniformis*, *Noctua armigera*, and *Noctua barbara*

Common Name(s)
Old world bollworm, scarce bordered straw worm, corn earworm, African cotton bollworm, American bollworm, tomato worm, gram pod borer

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description
Eggs: Freshly laid eggs (Fig. 1A) are hemispherical to spherical in shape, 0.4 to 0.6 mm in diameter with a flat base, and yellowish-white in color; changing to a deep yellow after a day. The eggs then change to dark or gray black a day before hatching (Bhatt and Patel, 2001; CABI, 2004). The eggs are sculpted with vertical ridges of alternating length, which surround a smooth apical area that contains the micropile (King, 1994).

Larvae: Larval color darkens with successive molts for the six instars typically observed for *H. armigera*. Coloration can vary considerably due to diet content. Coloration ranges from bluish green to brownish red (Fowler and Lakin, 2001). Freshly emerged first instar larvae are translucent and yellowish-white in color with a black to brown head capsule and have a spotted appearance (Fig. 1B) due to sclerotized setae, tubercle bases and spiracles (King, 1994; Bhatt and Patel, 2001). Second instar larvae are yellowish green in color with black thoracic legs. The full grown larvae are brownish or pale green with brown lateral stripes and distinct dorsal stripe; long and ventrally flattened but convex dorsally. Larval size in the final instar ranges from 3.5 to 4.2 cm in length (King, 1994).

Pupae: Dark-brown, 14 to 22 mm long and 4.5 to 6.5 mm in width, with a smooth surface, rounded both anteriorly and posteriorly, with two tapering parallel spines at posterior tip.

Adults: Stout-bodied moth of typical noctuid appearance (Fig. 1C), with 3.5 to 4 cm wing span; broad across the thorax and then tapering, 18 to 19 mm long. The coloration varies from dull greenish yellow to olive gray or light brown and females are darker than males (King, 1994).

For more information on descriptions see Common (1953), Kirkpatrick (1961), Hardwick (1965, 1970), King (1994).

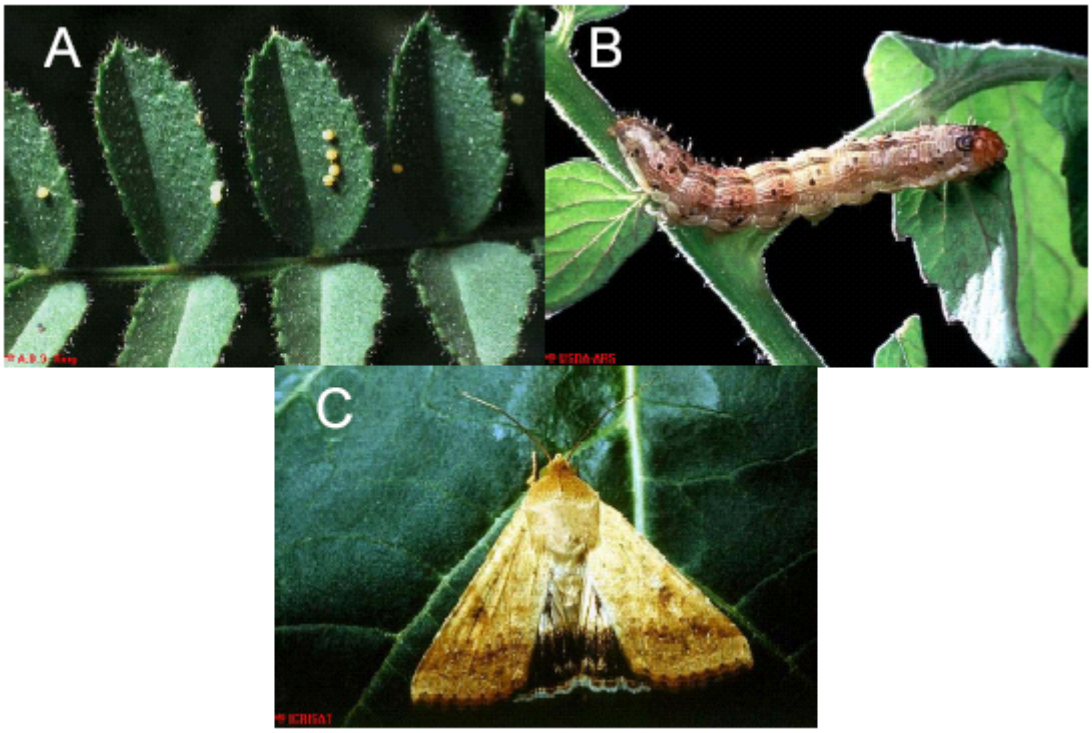

Figure 1. Life stages of *Helicoverpa armigera* (images not to scale): (A) eggs, (B) larva, and (C) adult. Photos courtesy of CABI, 2004.

Biology and Ecology (From Venette et al., 2003)
Because *H. armigera* exhibits overlapping generations, it can be difficult to determine the number of completed generations. Typically 2-5 generations are achieved in subtropical and temperate regions and up to 11 generations can occur under optimal conditions, particularly in tropical areas (Tripathi and Singh, 1991; King, 1994, Fowler and Lakin, 2001). Temperature and availability of suitable host plants are the most important factors influencing the seasonality, number of generations, and the size of *H. armigera* populations (King, 1994).

Adults emerge from the ground in the spring between dusk and midnight, climb vertical structures, and dry their wings for a period of 2 or more hours (King, 1994; CABI, 2004). In order to mate and lay eggs, adults typically must feed on nectar. About 2-5 days after emergence, females release a pheromone during early morning hours before dawn to attract mates (King, 1994). Mating occurs 1-4 days after emergence and is strongly influenced by humidity and temperature (King, 1994; Saito, 1999; Fowler and Lakin, 2001).

H. armigera lays eggs prolifically (Tripathi and Singh, 1991). A female may produce a maximum of 4394 eggs, but on average a female will produce 730-1,702 eggs (King, 1994; Fowler and Lakin, 2001; CABI, 2004). Eggs can be laid over 10 to 23 days (King, 1994). Oviposition begins 2-6 days after emergence, and egg laying often occurs at night (Kyi and Zalucki, 1991; Akashe et al., 1997; Fowler and Lakin, 2001; CABI 2004). Moths tend to lay eggs singly, on or near floral structures. Peak egg-laying typically occurs prior to or during host flower production (King, 1994). Depending on the quality of the host, *H. armigera* may also lay eggs on leaf surfaces. Female moths tend to choose pubescent (hairy) surfaces for oviposition rather than smooth leaf surfaces (King, 1994). In India, heavy egg-laying is a normal occurrence after rainfall (Das et al., 2001). In the Sudan, there is a significant rise in egg numbers after 6 days had elapsed following a rainfall event (Madden et al., 1993). King (1994) reviewed several adult longevity studies and reports a range in adult life span of 5 to 36 days.

Under adverse conditions, moths can migrate long distances (King, 1994; Zhou et al. 2000, Casimero et al., 2001; Shimizu and Fujisaki, 2002; CABI, 2004). Adults can disperse distances of 10 km during “non-migratory flights” and hundreds of kilometers (up to 250 km) when making “migratory flights”, which probably occur when host quality or availability declines (Saito, 1999; Zhou et al.; 2000, Casimero et al., 2001; Fowler and Lakin, 2001).

Eggs hatch in about 3 days at 25°C, but at lower temperatures, hatch may take up to 11 days. Larvae may complete up to 7 instars, though generally there are between 5 and 7 instars (Twine; 1978; King, 1994; Fowler and Lakin, 2001). In laboratory studies, the complete larval period (all instars combined) lasted between 12-36 days (Kirkpatrick, 1962; Bhatt and Patel, 2001; Fowler and Lakin, 2001). During summer, larval development is completed in 14-18 days, while it may take up to 21 days in fall (CABI, 2004). The prepupal stage lasts 1-4 days, and during this time larval activity decreases (King, 1994).

Molting often occurs in full sun on leaf surfaces (King, 1994). Before feeding on their host plant, newly hatched larvae typically consume all or part of their egg shells; larvae may then feed on leaf surfaces or floral structures, moving about the plant for a short distance before selecting a preferred feeding spot (King, 1994). Small, young larvae have the ability to feed inside floral structures, detectable only by a small hole with spun silk at the entrance and visible frass; larger larvae feed with a portion of their body outside the floral or fruiting structure (King, 1994).

Helicoverpa armigera is particularly damaging to crops because larvae can move from plant to plant, particularly when food is scarce (King, 1994). Late-instar larvae are more damaging to the host plant due to their attraction to “full buds” (Mabbett et al., 1980).

Once feeding is completed, larvae move 2.5-17.5 cm below the soil surface to pupate (King, 1994). Less frequently, pupation occurs within a spun web on the host plant (*e.g.*, in a corn cob) or on the soil surface (King, 1994). Depending on temperature, the pupal stage lasts between 6-33 days, unless the insect goes into diapause, in which case pupation may require several months. *H. armigera* overwinter as pupae (King, 1994; Akashe et al., 1997; Maelzer and Zalucki, 1999, Bhatt and Patel, 2001; Fowler and Lakin, 2001; CABI, 2004).

Diapause is facultative and occurs during the pupal stage (King, 1994). Diapause induction begins when larvae are exposed to day lengths between 11.5-12.5 hours, and low temperatures (19-23°C), or when larvae are exposed to lengthy periods of extremely hot and dry weather (≥ 35°C) (King, 1994; Zhou et al., 2000). Little to no diapause occurs in tropical areas (King, 1994). Total longevity (from egg to adult death) is 30-40 days with females generally living 2-3 days longer than males (King, 1994; Akashe et al., 1997). Bhatt and Patel (2001) recorded a slightly longer life span of about 51 days for males and 54 days for females. Rochester et al. (2002) reported a span 35-75 days from egg to adult.

The optimum temperature for development from 1st instar larva to adult was 33.9°C (Twine 1978) when reared on artificial diet. However, Twine (1978) reported optimal survival temperatures of 27° C for pupae and 24° C for larvae. In a laboratory study, high temperatures (above 37°C) caused pupal dormancy (Nibouche, 1998). A standard threshold for development of *H. armigera* was determined to be 11°C (Twine, 1978; Maelzer and Zalucki, 1999).

Symptoms/Signs

In sorghum and other grains, the larvae feed on the head when the grains are in the milky stage. They are especially damaging to sorghum varieties with tight compact heads. Varieties with loose open panicles are rarely damaged (Bijlmakers, 1989). Yield loss is caused by *H. armigera* feeding directly on the grain.

In other host species, bore holes are visible at the base of flower buds, the latter being hollowed out. Leaves and shoots may also be consumed by larvae. Larger larvae bore into maturing flowers, fruit, and seed. It may be necessary to cut open the plant organs to detect the pest. Frass may be evident. Fruit drop and defoliation are possible. Secondary infections by other organisms are common and lead to rotting.

Pest Importance

Heliothine moths of the genus *Helicoverpa* are considered to be among the most damaging insect pests in Australian agriculture, costing approximately $225.2 million per year to control (Clearly et al., 2006). *H. armigera* is a major insect pest of both field and horticultural crops in many parts of the world (Fitt, 1989). The pest status of *H. armigera* is due in part to the highly polyphagous nature of its larvae, its high fecundity, its high mobility, and its abilty to enter facultative diapause (Cleary et al., 2006). These characteristics make *H. armigera* particularly well adapted to exploit transient habitats such as man-made ecosystems.

Worldwide, *H. armigera* has been reported on over 180 cultivated hosts and wild species in at least 45 plant families (Venette et al., 2003). The larvae feed mainly on the flowers and fruit of high value crops and thus high economic damage can be caused at low population densities (Cameron, 1989; CABI, 2004). *H. armigera* is capable of long-distance migratory flights (King, 1994; Zhou et al., 2000; Casimero et al., 2001; Shimizu and Fujisaki, 2002; CABI, 2004).

Management of *Helicoverpa* spp. in the past has relied heavily on the use of insecticides, and this has led to resistance problems in cotton (Fitt, 1994). Resistance to pyrethroids amongst *H. armigera* is a serious problem (McCaffrey et al., 1989; Trowell et al., 1993).

Known Hosts

Major hosts

Abelmoschus esculentus (okra), *Allium* spp. (onions, garlic, leek, etc.), *Arachis hypogaea* (peanut), *Avena sativa* (oats), Brassicaeae (cruciferous crops), *Cajanus cajan* (pigeon pea), *Capsicum annuum* (bell pepper), *Carthamus tinctorius* (safflower), *Cicer arietinum* (chickpea, gram), *Citrus*, Cucurbitaceae (cucurbits), *Diathus caryophyllus* (carnation), *Eleusine coracana* (finger millet) *Glycine max* (soybean), *Gossypium* (cotton), *Helianthus annuus* (common sunflower), *Hordeum vulgare* (barley), *Lablab purpureus* (hyacinth bean), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Malus* spp. (apple), *Mangifera indica* (mango), *Medicago sativa* (alfalfa), *Nicotiana tabacum* (tobacco), *Pennisetum glaucum* (pearl millet), *Phaseolus* (beans), *Phaseolus vulgaris* (common bean), *Pinus* spp.(pines), *Pisum sativum* (pea), *Prunus* spp. (stone fruit), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Triticum* (wheat), *Triticum aestivum* (wheat), *Vigna unguiculata* (cowpea), and *Zea mays* (maize) (CABI, 2004).

Wheat, oats, and barley are considered primary hosts for *H. armigera.*

Note: Not all host plants are equally preferred for oviposition but can be utilized in the absence of a preferred host. There have been several studies within the laboratory setting on host preference. Jallow and Zalucki (1996) found that most females ranked maize, sorghum, and tobacco highest, followed by cotton

varieties. The least preferred were cowpea and alfalfa. Pigeonpea and maize are considered to be the most suitable host for this insect, when compared to sorghum, red ambadi (*Hibiscus subdariffa*), marigold, and artificial diet (Bantewad and Sarode, 2000). Tobacco, maize, and sunflower were categorized as the most preferred hosts; soybean, cotton, and alfalfa were categorized as intermediate hosts; and cabbage, pigweed, and linseed were the least preferred in an additional study (Firempong and Zalucki, 1990).

Poor hosts
Vitis vinifera (grape) (Vorus, 1996).

Wild hosts
Acalypha spp. (copperleaf), *Amaranthus* spp. (pigweed, amaranth), *Datura* spp., *Datura metel* (datura), *Gomphrena*, *Hyoscyamus niger* (black henbane), Sonchus oleraceus (annual sowthislte) (Gu and Walter, 1999; CABI, 2004).

For a complete listing of hosts see Venette (2003).

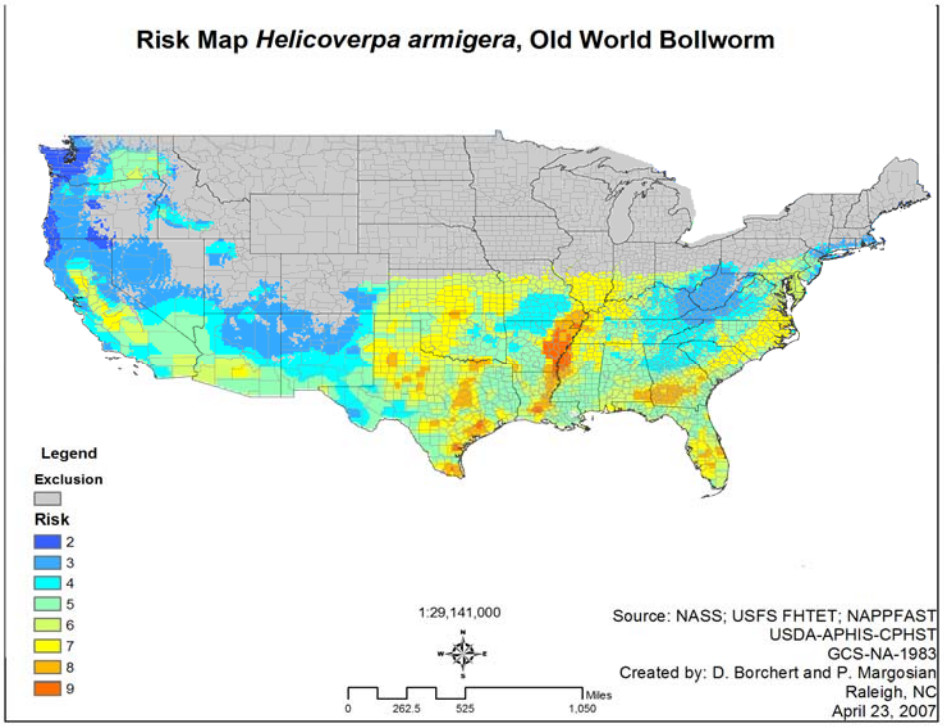

Figure 2. Risk map for *H. armigera* within the continental United States. The greater the risk (climate and host availability), the greater the risk number. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Known Vectors (or associated organisms)

Helicoverpa armigera is not a known vector and does not have any associated organisms.

Known Distribution

Helicoverpa armigera is found in the Palearctic, Oriental, Ethiopian, and Australian zoogeographic provinces, south of a line at approximately 52°N. The range occupied by the species includes tropical, dry, and temperate climates (CABI, 2004).

Asia: Afghanistan, Armenia, Azerbaijan, Bhutan, Brunei Darussalam, Cambodia, China, Cocos Islands, Republic of Georgia, India, Indonesia, Iran, Iraq, Israel, Japan, Jordan, Kazakhstan, Korea, Kuwait, Kyrgyzstan, Laos, Lebanon, Malaysia, Myanmar, Nepal, Philippines, Saudi Arabia, Singapore, Sri Lanka, Syria, Tajikistan, Thailand, Turkey, Turkmenistan, United Arab Emirates, Uzbekistan, Vietnam, Yemen. **Europe:** Albania, Bulgaria, Cyprus, Finland, France, Germany, Greece, Hungary, Italy, Macedonia, Malta, Portugal, Romania, Russian Federation, Serbia and Montenegro, Slovenia, Spain, Switzerland, Ukraine. **Africa:** Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Congo, Congo, Cote d'Ivoire, Egypt, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Lesotho, Libya, Madagascar, Malawi, Mauritania, Mauritius, Mayotte, Morocco, Mozambique, Namibia, Niger, Nigeria, Reunion, Rwanda, Saint Helena, Senegal, Seychelles, Sierra Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zambia, Zimbabwe. **Oceania:** American Samoa, Australia, Belau, Federated States of Micronesia, Fiji, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Norfolk Island, Northern Mariana Islands, Papua New Guinea, Samoa, Solomon Islands, Tonga, Tuvalu, and Vanuatu.

Potential Distribution Within the United States

According to Fowler and Lakin (2001), it is probable that *H. armigera* could establish in every state in the continental U.S. based on habitat and host suitability. *Helicoverpa armigera* would probably pose the greatest economic threat to the following states: Alabama, Arizona, Arkansas, California, Georgia, Illinois, Iowa, Kansas, Louisiana, Michigan, Minnesota, Mississippi, Nebraska, New Mexico, North Carolina, Ohio, Pennsylvania, South Carolina, South Dakota, Tennessee, Texas, Virginia, and Wisconsin. A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 2) indicates that the southern United States has the greatest risk for *H. armigera* establishment based on host availability and climate within the continental United States.

Survey

Preferred Method: **(From Venette et al., 2003).** Pheromone traps using (*Z*)-11-hexadecenal and (*Z*)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera* (Pawar et al., 1988; Loganathan and Uthamasamy,

1998; Loganathan et al., 1999; Visalakshmi et al., 2000; Zhou et al., 2000). Of three pheromone doses tested in the field (0.75, 1.0, and 1.25 mg/septum), 1 mg attracted the most males (Loganathan and Uthamasamy, 1998); the trap type was not specified. Rubber septa impregnated with these sex pheromone components (1 mg/septum) were equally effective in capturing males for 11 days in the laboratory (Loganathan et al., 1999). Captures of *H. armigera* in the field were significantly lower with 15-day-old lures than with fresh lures, and the authors recommend replacing lures every 13 days (Loganathan et al., 1999). Similar observations were reported by Pawar et al. (1988). Males responded to the pheromone during dark hours only, commencing at 1800 hour and terminating at 0600 hour. The highest response was between 2300 and 0400 hours (Kant et al., 199).

Trap design has a significant impact on the number of male *H. armigera* moths that will be captured with pheromone lures. Funnel traps and Texas traps are substantially more effective than sticky traps (Kant et al., 1999). Hartstack *(i.e.,* hollow cone) traps have also been used to effectively monitor densities of adults (Walker and Cameron, 1990). Cone traps are significantly more effective than water-pan traps (Sheng et al., 2002). Traps should be placed approximately 6 feet (1.8 meter) above the ground (Kant et al., 1999; Zhou et al., 2000), and they should be separated by a distance of at least 160 feet (50 meters) (Kant et al., 1999). For routine monitoring of pests, pheromone traps are deployed at a density of 5 traps/hectare (Sidde Gowda et al., 2002).

Adults of both sexes can be captured in black light traps.

Alternative Method: Visual inspections of plants for eggs and/or larvae are frequently used to monitor and assess population sizes for *H. armigera*. Females lay several hundred eggs on all parts of the plant, flowers and fruits. Eggs may hatch in less than 3 days at an optimum temperature of 27 to 28°C. The feeding larvae can be seen on the surface of plants but they are often hidden within plant organs (flowers, fruits, etc.). Bore holes and heaps of frass (excrement) may be visible, but otherwise it is necessary to cut open the plant organs to detect the pest. Because larvae are available for sampling only a short time, some surveys have been conducted by only sampling damaged fruit (Bouchard et al., 1992). In temperate regions, *H. armigera* overwinters as a pupa buried several cm in the soil. Adults appear in April to May and can be observed until October, because of the long migration period.

In vegetative Australian cotton, a minimum of 60 whole plants per 100 hectare commercial field are examined for the presence of *H. armigera* eggs or larvae; when plants begin to produce squares, only the upper terminal (approximately 20 cm) of a plant is inspected (Brown, 1984; Dillon and Fitt, 1995). In experimental plots, visual inspections for *H. armigera* in pigeon pea were restricted to the upper third of whole plants (4 sets of five plants in a 30 x 30 meter plot) (Sigsgaard and Ersbøll, 1999).

Leaves of tomato plants are more attractive than flowers or fruits as *H. armigera* oviposition sites, but use of a single-leaf sample unit (with a sample size of 30 plants per field) has proven ineffective in detecting low densities of *H. armigera* (Cameron et al., 2001). On some tomato cultivars, leaves in the upper half of the plant are preferentially selected for oviposition (Saour and Causse, 1993).

Key Diagnostics

Adults *H. armigera* may be identified by distinct differences in genitalia (Kirkpatrick, 1961; Hardwick, 1965). Differentiation between *H. armigera* and *H. zea* is very difficult; identification is by dissection of internal structures of adult males (Pogue, 2004). A morphological study of *H. assulta, H. punctigera,* and *Heliothis virescens* (formerly *H. rubrescens*) compares similarities and differences between species; a key is provided for identifying adults (Kirkpatrick, 1961). Immunological tests are available to differentiate *H. punctigera* and *Heliothis virescens* in egg or larval stages (Ng et al., 1998).

The LepTon test, an Enzyme Linked Immunosorbent Assay (ELISA) based approach, has been developed to distinguish between *H. armigera* and *H. punctigera* in all stages (Trowell et al., 1993).

Agusti et al. (1999) developed sequence amplified characterized region (SCAR) markers to detect *H. armigera* eggs in the gut of predators.

Easily Confused Pests

Several noctuid pests can be confused easily with *H. armigera*, including *H. assulta* (not known in the United States)*, H. punctigera (*not known in the United States)*, H. zea* (present in the United States)*,* and *Heliothis virescens* (present in the United States) (Kirkpatrick, 1961; CABI, 2004).

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Nysius huttoni

Scientific Name
Nysius huttoni White

Synonyms:

Common Name(s)
Wheat bug, wheat seed bug, New Zealand wheat bug, Nysius bug

Type of Pest
True bug

Taxonomic Position
Class: Insecta, **Order:** Hemiptera, **Family:** Lygaeidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe (2009)

Pest Description
Notes on taxonomy: *Nysius huttoni* is an extremely variable species, with three inter-breeding forms based on the extent of wing development (Aukema et al., 2005).

Eggs: Oval, length about three times width; mean length about 0.8 mm, mean width about 0.3 mm. Straw yellow to creamy white; cephalic (head) end more orange when first laid, deep orange when about to hatch (Eyles, 1960).

Nymphs: Generally pale gray to orange (Fig. 1), marked with varying degrees of brown, black, and gray; length from about 0.5 mm in instar I to about 2.0 mm in instar V. Head dark brown to black with longitudinal pale gray to orange stripes. Instars I-IV with pronutum (and wing pads in instars III-IV) dark brown to black; in instar V, pronutum pinkish to gray, variably marked with brown and black, lateral margins and mesal line pale, apex of wing pads and broad U-shaped mark on pronotum black. Dorsal surface of abdomen grayish blue, each segment with transverse row of whitish spots surrounded by narrow red ring. Legs pale brown, spotted with black (Eyles,

Figure 1. Nymph of *N. huttoni*. Photo courtesy of HortNET , The Horticulture and Food Research Institute of New Zealand. www.bugwood.org

1960).

Adults: (Fig. 2) Length 3.5-4.3 mm; width 1.3-1.8 mm. Dorsally clothed with short, appressed, golden to silvery, sericeous pubescence, intermixed with erect, simple setae. Head wider than long, black, mesal area yellow to reddish yellow. Antennae about twice as long as head width, brown to black, 1st segment sometimes yellowish. Pronutum trapeziform, distinctly punctuate, brown, humeral angles and base of meson yellow; scutellum shiny black. Hemelytra brown, variably mottled and spotted with yellow, corial margins uniformly brown, apical margin of each corium bordering the membrane with three dark-brown spots sometimes coalesced into extensive dark area. Membrane nearly clear, cross-hatched with white lines, basal area often fuscous. Undersurface mostly blak, abdomen mottled with yellow, coxal clefts yellow. Femora dark brown with apices, dorsal line, and edges broken by yellow, tibiae yellow, tarsi and claws yellow to brown (USDA, 1985).

Figure 2. Adult *N. huttoni*. Photo courtesy of Natasha Wright, Florida Department of Agriculture and Consumer Services, www.bugwood.org in

Adults can be divided into three size groups: large (3.5-4.3 mm), medium-sized (3.0-3.8mm), and small (2.3-3.2 mm). All individuals of the large group are long winged or macropterous, but adults the medium-sized and small groups have three forms ranging from macropterous, sub-bracypterous, to brachypterous (short winged) (Eyless, 1960).

Biology and Ecology

Nysius huttoni usually overwinters as an adult and undergoes reproductive diapause, which is induced by shortening daylengths during the late summer (Farrell and Stufkens, 1993). In lowland agricultural areas, the wheat bug overwinters as an adult under grasses and vegetable debris (Gurr, 1957). Early in the season this pest is associated with weeds, but the bug moves to wheat as most weeds mature and wheat reaches the milk-ripe stage. Adults appear in large numbers duing the summer on clover and other plants near wheat crops. *Nysius huttoni* also attacks seedling Brassica crops, which provide the ideal open ground cover. Adults thrive under hot, dry conditions, preferring situations wher sunlight reaches the ground. They are seldom found in dense vegetation. Adults hide under clods or debris on the ground when the temperature begins to fall in the evening and become active in the morning when the temperature rises. Rain inhibits activity (Gurr, 1957; USDA, 1985).

Mating occurs during the summer, with a single copulation fertilizing the female for life. One female may deposit singly or in clusters 25-248 eggs, usually in the cracks in the soil (Schaefer and Panizzi, 2000). Females may occasionally deposit eggs on host plants (He and Wang, 2000). Eggs hatch in about 10 days. Nymphs undergo five instars. The complete life cycle takes 50-65 days. There are at least three, possibly four, generations per year in New Zealand and two generations per year in the Netherlands and Belgium (Ferro, 1976; Aukema et al., 2005).

Long day lengths (16:8 and 14:10 h light:dark) promote a continuous lifecycle while short daylengths (12:12 and 10:14 h light:dark) slow up growth and development, promote the pre-mating period, and induce reproductive diapause (He et al., 2004). *N. huttoni* required 638.2, 648.88, and 637.21 degree days for the completeion of its lifecycle at 20, 25, and 30 °C, resepectively (He et al., 2003).

Symptoms/Signs

The wheat bug is a seed feeder, but may also feed on the foliage. Wheat is most vulnerable to damage at the flowering and grain filling stages of growth (Every et al., 1990). The bug attacks wheat kernels in the water ripe to milky ripe stages of development, piercing the grain and sucking out the juices. Damage usually occurs at field edges (USDA, 1985). When the bug-damaged grain matures, it is characterized by a dark insect-feeding puncture mark surrounded by a pale area on the surface. Some grains shrink to a cuboid shape when much endosperm is removed, presumably from prolonged feeding (Gurr, 1957). Seed germination is not affected by *N. huttoni.*

Because the wheat bug injects an enzyme during feeding, flour from damaged wheat ruins dough during breadmaking. The enzyme splits adjacent protein chains in dough, breaking down the dough structure, suddenly turning it runny and sticky. As few as three to four damaged grains per 1,000 produce flour unsuited for baking (Meredith, 1970).

Nysius huttoni is a particular problem on direct drilled brassica crops. On *Brassica* spp., stems, petioles, and leaves are preferred for feeding. Damage to crucifer seedlings appears throughout the field. A cankerous growth, induced by feeding punctures, rings the stems at the ground level. The girdled seedlings collapse on their own or break in high winds (Gurr, 1957; USDA, 1985).

Nysius huttoni has been reported to reduce seed fill and see quality in white clover.

Pest Importance

The genus *Nysius* occurs in all biogeographic regions of the world, and has many species attacking agricultural and horticultural crops. The New Zealand native bug, *Nysius huttoni*, has a wide host range, including a variety of crops and

weeds. *N. huttoni* is an important pest of wheat and brassica crops. The feeding of *N. huttoni* reduces the weight of grain but more importantly, adversely affects the flour. *N. huttoni* is also a serious pest of young *Brassica* spp; its feeding eventually leads to seedling death ending in poor crop stands (USDA, 1985). Damage is often severe especially in dry districts and elsewhere in dry years when serious outbreaks occur (Gurr, 1957; USDA, 1985).

The worst outbreak on wheat was recorded in 1970. About 10,000 tons of wheat were damaged by *N. huttoni* (Swallow and Cressey, 1987). According to Ferguson (1994), up to 70% of immature rutabaga (swede) (*B. napus rapifera*) plants were lost through wind breakage after attack by *N. huttoni.*

N. huttoni injects a salivary proteinase into immature wheat kernals while feeding. This proteinase remains in flour made from bug-damaged wheat and, in dough, digests gluten to produce slack, sticky dough and poor quality bread (Every et al., 2005).

Furthermore, *N. huttoni* is often found as a hitchking pest in apple packages for export from New Zealand (Birtles et al, 1992; Lay Yee et al., 1997). The high tolerance of late instar nymphs and adults of *N. huttoni* to low temperatures (10 and 15°C) allows the pest to survive lengthy shipment times in infested fruit packages, although the pest could not complete its lifecycle at these temperatures. These features of *N. huttoni* raise major quarantine risks to countries that trade with New Zealand (He et al., 2003).

Known Hosts

Major hosts

Brassica napus (rapeseed, swede), *Brassica napus* var. *napobrassica* (rutabaga, swede), *Brassica oleracea* (broccoli), *Brassica rapa* (turnip), *Brassica* spp. (mustard), *Trifolium dubium* (small hop clover), *Trifolium pratense* (red clover), *Trifolium repens* (white clover), *Trifolium subterraneum* (subterranean clover), *Triticum aestivum* (wheat), and *Triticum* spp. (wheat).

Other hosts:

Aciphylla scott-thomsonii (giant Spaniard speargrass), *Agrostis capillaria* (bent grass), *Avena sativa* (oat), *Bromus* spp. (bromegrass), *Chionochloa rubra* (red tussock), *Hebe salicfolia* (koromiko), *Helianthus annus* (common sunflower), *Hordeum sativum* (barley), *Lactuca* spp. (lettuce), *Lathyrus* spp. (sweet, perennial pea), *Linum* spp. (flax), *Lolium* spp. (ryegrass), *Medicago sativa* (alfalfa), *Pinus radiata* (Monterey pine), *Poa caespitosa* (silver tussock), *Poa colensoi* (blue tussock), *Pyrus pyranica, Secale cereale* (rye), and *Spergularia rubra* (red sandspurry).

Weed hosts:
Anagallis arvensis (scarlet pimpernel), *Achillea millefolium* (common yarrow), *Calandrinia* spp. (redmaid), *Capsella bursa-pastoris* (shepherd's purse), *Cassinia leptophylla* (tauhinu), *Chenopodium album* (common lambs quarters), *Coronopus didymus* (lesser swinecress), *Cytisus scoparius* (scotchbroom), *Hieracium* spp. (hawkweeds), *Nassella trichotoma* (serrated tussock), *Polygonum aviculare* (prostrate knotweed), *Polygonum maculosa*, *Rumex acetosella* (red sorrel), *Senecio inaequidens*, *Silene gallica* (English catchfly), *Soliva sessilis* (lawn burweed), *Stellaria media* (common chickweed), *Ulex europaeus* (common gorse), and *Verbascum thapsus* (common mullein).

It is also suggested that the presence of mosses (e.g. *Ceratodon, Sphagnum,* and *Polytrichum* spp.) may be crucial for the overwintering period.

Contaminant in:
Actinidia deliciosa (kiwi), *Fragaria* x *ananassa* (strawberry), *Malus domestica* (apple), and *Rubus* spp. (blackberry, raspberry).

Gurr (1957), Ferro (1976) and Scott (1984) suggest that strawberry and raspberry are also attacked but Farrell and Stufkens (1993) consider the presence of adults on strawberries and kiwifruit to be simply a contamination problem. It is stated that it is not a pest of apples although it has been found in consignments (Birtles *et al*., 1992).

Known Vectors (or associated organisms)
Nysius huttoni is not a known vector and does not have any associated organisms.

Known Distribution
Europe: Belgium, Netherlands. **Oceania:** New Zealand

Potential Distribution Within the United States
Nysius huttoni can move as nymphs and adults in commercial shipments of imported fruits and vegetables. In the Netherlands, *N. huttoni* has become extremely abundant raising concerns that this pest may be transported to the United States through trade. *Nysius huttoni* has been intercepted numerous times on plant species not recorded as hosts, which creates additional concern about this pest.

Research in New Zealand indicates that *N. huttoni* could establish in countries that experience temperatures higher than 15°C with a maximum population development occurring at 20°C to 30°C (He et al., 2003). The current distribution of *N. huttoni* in the Netherlands corresponds to U.S. plant hardiness zone 8 (Appendix E), indicating that *N. huttoni* is capable of establishing in areas with an average minimum temperature of -12.2°C (10°F) (NPAG, 2006). Zone 8 includes

areas of Washington, Oregon, California, Nevada, Arizona, New Mexico, Texas, Louisianna, Arkansas, Mississippi, Alabama, Georgia, Florida, South Carolina, and North Carolina. However, recent finds in Belgium and Netherlands suggest that zones 6 and 7 are also at risk. Zones 6 and 7 include a number of wheat producing states. A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 3) indicates that the southern United States has the greatest risk for *N. huttoni* establishment based on host availability and climate within the continental United States, but many wheat growing areas of the United States are at considerable risk.

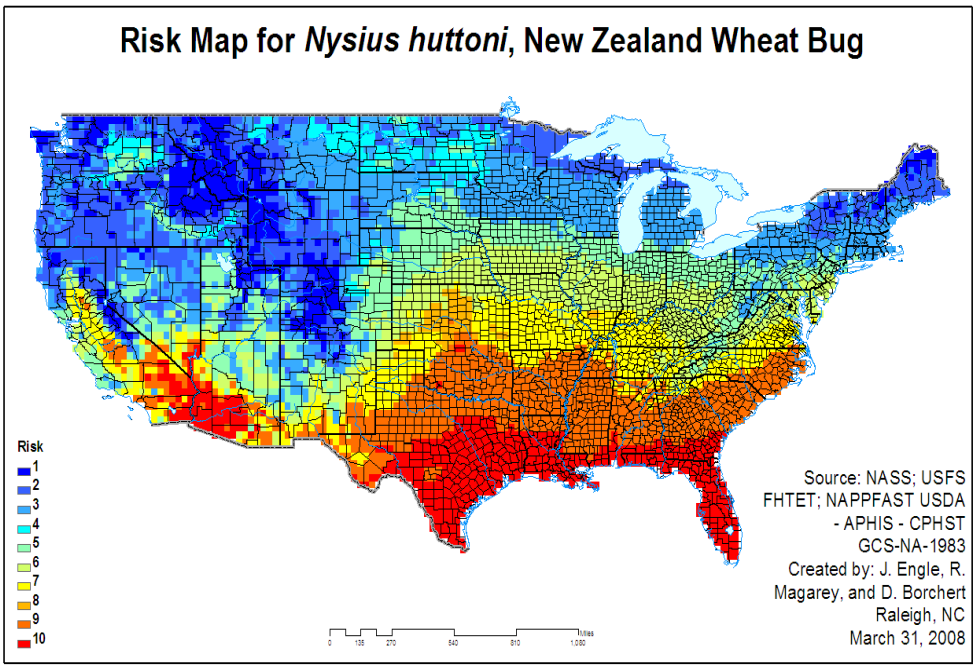

Figure 3. Risk map for *N. huttoni* within the continental United States. The greater the risk (climate and host availability), the greater the risk number. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Survey

Preferred Method: Survey is primarily conducted by visual examination of wheat. The bug attacks wheat kernels in the water ripe to milky ripe stages of development, piercing the grain and sucking out the juices, thus surveys should be conducted at this time. Damage usually occurs at field edges (USDA, 1985).

Bug damage to mature wheat kernels can be easily recognized as pale, slightly elevated patches, often with one or more black or red dots considered to be the marks of bug stylet punctures, or pitting, blackening, and distortion (especially under the microscope) (Every et al., 1998).

In wheat, different cultivars appear to be more susceptible to *N. huttoni* damage. In general, it appears that wheat of high bread quality, such as Otane, Oroua, Domino, and Batten, are less susceptible that poor bread quality wheat varieties, such as Karamu, WE378, ASPS9927, and ASPS9928 (Every et al., 1998).
In the Netherlands, *N. huttoni* is primarily found in areas with sparse vegetation surrounded by bare ground, such as in waste sites and other disturbed locations and not in crop fields. This may be typical of an early introduction and some time should be devoted to these areas.These sites contain an abundance of moss, which has led to speculation that moss may be necessary of overwintering in the Netherlands (NPAG, 2006).

Alternative Methods: The following methods have been utilized when the wheat bug was known to be present to quantify populations. Soil to a depth of 10 mm plus all plant material was removed from each quadrant (8 randomly placed 0.25m^2 quadrants), bagged, and taken to the laboratory (Farrell and Stufkens, 1993). After agitation in a 25% saline solution and removing the floatant, the retained material was examined under a microscope. All wheat bug development stages were identified.

A flight trap was set up 1-2 m above the ground. The trap comprised two plates of 3 mm thick Perspex each 200 mm x 800 mm, interlocked to form a cruciform section baffle 800 mm high with four vanes projecting 100 mm from the center line. The was mounted over a 200 mm diameter funnel to receive the catch, opening into a collecing jar containing kerosene (Farrell and Stufkens, 1993). Additionally, wheat bugs flying at approximately 7.5 m above the ground were trapped in an enclosed suction trap equipped with a 300 mm diameter 2800 rpm aerofoil fan.

Key Diagnostics
Submit adults for identification in alcohol or mounted dry on triangular points (USDA, 1985).

Easily Confused Pests
Information is not currently available.

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Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

Synonyms:
Hadena littoralis, Noctua gossypii, Prodenia littoralis, Prodenia litura, Prodenia retina, Prodenia retina, Prodenia testaceoides

The two Old World cotton leafworm species *S. littoralis* and *S. litura* are allopatric, their ranges covering Africa and Asia, respectively. Many authors have regarded them as the same species.

Common Name(s)
Cotton leafworm, Egyptian cotton leafworm, Mediterranean climbing cutworm, tobacco caterpillar, tomato caterpillar, Egyptian cotton worm, Mediterranean brocade moth, Mediterranean climbing cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description **(From CABI, 2004)**
Eggs: Spherical, somewhat flattened, 0.6 mm in diameter, laid in clusters arranged in more or less regular rows in one to three layers, with hair scales derived from the tip of the abdomen of the female moth (Fig. 1). The hair scales give the eggs a “felt-like appearance”. Usually whitish-yellow in color, changing to black just prior to hatching, due to the big head of the larva showing through the transparent shell (Pinhey, 1975).

Larvae: Upon hatching, larvae are 2-3 mm long with white bodies and black heads and are very difficult to detect visually**.** Larvae grow to 40 to 45 mm and are hairless,

Figure 1. Eggs and neonates. Eggs are laid in batches covered with orange-brown hair scales. Photo courtesy of http://www.defra.gov.uk/planth/pestnote/spod.htm

cylindrical, tapering towards the posterior and variable in color (blackish-gray to dark green, becoming reddish-brown or whitish-yellow) (Fig. 2). The sides of the body have dark and light longitudinal bands; dorsal side with two dark semilunar spots laterally on each segment, except for the prothorax; spots on the first and eighth abdominal segments larger than the others, interrupting the lateral lines on the first segment. The larva of *S. littoralis* is figured by Bishari (1934) and Brown and Dewhurst (1975). Larvae are nocturnal and during the day can be found at the base of the plants or under pots.

Figure 2. Larva of *S. littoralis*. Photos courtesy of CABI, 2004.

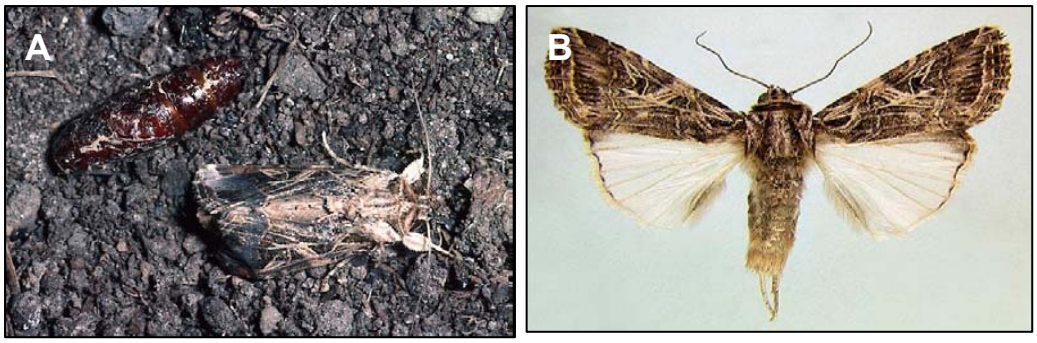

Figure 3. Pupa and adult of *S. littoralis* on soil (A). Adult moth of *S. littoralis* (museum set specimen) (B). Photos courtesy of CABI, 2004 and Entopix.

Pupae: When newly formed, pupae are green with a reddish color on the abdomen, turning dark reddish-brown after a few hours (Fig. 3A). The general shape is cylindrical, 14-20 x 5 mm, tapering towards the posterior segments of the abdomen. The last segment ends in two strong straight hooks (Pinhey, 1975).

Adults: Moth with gray-brown body (Fig. 3B), 15 to 20 mm long; wingspan 30 to 38 mm; forewings gray to reddish brown with paler lines along the veins (in males, bluish areas occur on the wing base and tip); the ocellus is marked by two or three oblique whitish stripes. Hindwings are grayish white, iridescent with gray margins and usually lack darker veins (EPPO, 1997).

Biology and Ecology

Spodoptera littoralis larvae damage many agricultural plants, particularly cotton (Venturini, 1975). Adult moths feed on nectar, and females oviposit eggs on the leaves of crop plants.

Depending on the climate of the region, *S. littoralis* can have from two to seven generations per year and does not undergo diapause (Salem and Salama, 1985). Egg masses consist of hundreds of eggs and are most abundant on young leaves on the upper parts of the plant (Khalifa et al., 1982), the undersurface of leaves (Nasr and Nassif, 1970; Gawaad and El Gayar, 1974), and the younger leaves (Khalifa et al., 1982).

As the moth develops, it completes six larval instars. First through third instars do not move about the plant, hence 80% of early instar larvae inhabit the same location where the eggs were deposited (*i.e.,* the upper parts of plants and the lower leaf surfaces) (Hoeny et al., 1982). Late instar larvae move about the plant and appear to prefer the upper parts of plants during the early morning hours and the lower plant areas or the soil during the afternoon hours. Both the early and late instars larvae avoid the mid-regions of the plants (Abdel Megeed and Iss Hak, 1975). Fourth through sixth instars move to the ground during the hot hours of the day, and late sixth instars bury themselves in the soil to pupate (Gawaad and El Gayar, 1974). Adult moths emerge at night and can live for 5-10 days (Salama and Shoukry, 1972). About half of females will lay their eggs before sunrise the same night of mating (Hassan et al., 1960).

Egg masses have shown to have variable distribution in fields. One study reported that the distribution egg masses where increased towards the center of the cotton field (Iss Hak and Abdel Megeed, 1975), whereas another study reported that in some years *S. littoralis* was more abundant at the edges and in other years it was more abundant in the center of the field (Khalifa et al., 1982).

The lower threshold temperatures for egg, larvae, pupae and pre-oviposition periods was 11.86, 7.69, 12.34 and 10.66 °C, respectively (Dahi, 2005). The upper temperature threshold for complete development of *S. littoralis* is 37 °C (El-Malki, 2000). *Spodoptera littoralis* requires 53.2, 314.7, 155.8 and 27.5 degree-days for egg, larvae, pupae and pre- oviposition period, respectively (Dahi, 2005). At temperatures of 18 °C and 36 °C, eggs hatched within 2 and 9 days, larval stage lasted 10 and 35 days, and pupal stage took 8 and 27 days, respectively (Ocete Rubio, 1984). The optimal temperature for maximum weight gain in larvae is 20 °C (Bhatt, 1976), egg production is 25 °C (Nasr, 1974), larval

survivorship to adult is 25 °C (Sidibea and Lauge, 1977; Hegazi and Schopf, 1984; Ocete Rubio, 1984), and pupation is 30 °C (Nasr and Nassif ,1977).

In general, cold resistance is the lowest in the egg stage, increases with in maturing larvae, and is greatest in the pupal stage (Miller, 1977).

In one study, most moths flew from up to 250 m to 500 m from release point, and the farthest recapture obtained was at 1500 m (Salama and Shoukry, 1972). In another study, most males were captured within 100 m and most were recaptured the same night as they were released (Kehat et al., 1976).

Pest Importance

Spodoptera littoralis is one of the most destructive agricultural lepidopterous pests within its subtropical and tropical range. The pest causes a variety of damage as a leaf feeder and sometimes as a cut worm on seedlings. It can attack numerous economically important crops throughout the year (EPPO, 1997). On cotton, the pest may cause considerable damage by feeding on the leaves, fruiting points, flower buds and occasionally on bolls. When peanuts are infested, larvae first select young folded leaves for feeding, but in severe attacks, leaves of any age are stripped off. Sometimes, even the ripening kernels in the pods in the soil may be attacked. Pods of cowpeas and the seeds they contain are also often badly damaged. In tomatoes, larvae bore into the fruit, rendering them unsuitable for consumption. Numerous other crops are attacked, mainly on their leaves.

In Europe, damage caused by *S. littoralis* was minimal until about 1937. In 1949, there was a catastrophic population explosion in southern Spain, which affected alfalfa, potatoes and other vegetable crops. At present, this noctuid pest is of great economic importance in Cyprus, Israel, Malta, Morocco and Spain (except the north). In Italy, it is especially important on protected crops of ornamentals and vegetables (Inserra and Calabretta, 1985; Nucifora, 1985). In Greece, *S. littoralis* causes slight damage in Crete on alfalfa and clover only. In North Africa, tomato, *Capsicum*, cotton, maize and other vegetables are affected. In Egypt, it is one of the most serious cotton pests (CABI, 2004).

Many populations of *S. littoralis* are extremely resistant to pesticides, and if they become well established, can be exceptionally difficult to control (USDA, 1982).

Symptoms/Signs

On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants (USDA, 1982). In some crops, fruiting points, flower buds, and fruit/seed are also damaged.

Known Hosts

The host range of *S. littoralis* covers over 40 families, containing at least 87 species of economic importance (Salama et al., 1971).

Major Hosts
Abelmoschus esculentus (okra), *Allium* spp. (onion), *Amaranthus* spp. (pigweed, amaranthus), *Apios* spp. (groundnut), *Arachis hypogea* (peanut), *Beta vulgaris* (beet), *Brassica oleracea* (cabbage, broccoli), *Brassica rapa* (turnip), *Brassica* spp. (mustards), *Camellia sinensis* (tea), *Capsicum annuum* (pepper), *Chrysanthemum* spp., *Citrullus lanatus* (watermelon), *Citrus* spp., *Coffea arabica* (coffee), *Colocasia esculenta* (taro), *Corchorus* spp. (jute), *Cucumis* spp. (squash, pumpkin), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Dianthus caryophyllus* (carnation), *Ficus* spp. (fig), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annus* (sunflower), *Ipomoea batatas* (sweet potato), *Lactuca sativa* (lettuce), *Linum* spp. (flax), *Lycopersicon esculentum* (tomato), *Medicago sativa* (alfalfa), *Morus* spp. (mulberry), *Musa* spp. (banana, plantain), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Pennisetum glaucum* (pearl millet), *Persea americana* (avocado), *Phaseolus* spp. (bean), *Pisum sativum* (pea), *Prunus domestica* (plum), *Psidium guajava* (guava), *Punica granatum* (pomegranate), *Raphanus sativus* (radish), *Rosa* spp. (rose), *Saccharum officinarum* (sugarcane), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Spinacia* spp. (spinach), *Theobroma cacao* (cacao), *Trifolium* spp. (clover), *Triticum aestivum* (wheat), *Vicia faba* (broad bean), *Vigna* spp. (cowpea, black-eyed pea), *Vitis vinifera* (grape), and *Zea mays* (corn).

Minor Hosts
Acacia spp. (wattles), *Actinidia arguta* (tara vine), *Alcea rosea* (hollyhock), *Anacardium occidentale* (cashew), *Anemone* spp. (anemone), *Antirrhinum* spp., *Apium graveolens* (celery), *Asparagus officinalis* (asparagus), *Caladium* spp. (caladium), *Canna* spp. (canna), *Casuarina equisetifolia* (she-oak), *Convolvulus* spp. (morning glory, bindweeds), *Cryptomeria* spp. (Japanese cedar), *Cupressus* spp. (cypress), Datura spp. (jimsonweed), *Eichhornia* spp. (waterhyacinth), *Eucalyptus* spp. (eucalyptus), *Geranium* spp. (geranium), *Gladiolus* spp. (gladiolus), *Malus domesticus* (apple), *Mentha* spp. (mint), *Phoenix dacylifera* (date palm), *Pinus* spp. (pine), and *Zinia* spp. (zinnia).

Known Vectors (or associated organisms)
Spodoptera littoralis is not a known vector and does not have any associated organisms.

Known Distribution
The northerly distribution limit of *S. littoralis* in Europe corresponds to the climatic zone in which winter frosts are infrequent. It occurs throughout Africa and extends eastwards into Turkey and north into eastern Spain, southern France and northern Italy (CABI, 2004). However, this boundary is probably the extent of migrant activity only; although the pest overwinters in southern Spain, it does not do so in northern Italy or France. In southern Greece, pupae have been observed in the soil after November and the species overwinters in this stage in Crete. Low

winter temperatures are, therefore, an important limiting factor affecting the northerly distribution, especially in a species with no known diapause (Miller, 1976; Sidibe and Lauge, 1977).

Africa: Algeria, Angola, Benin, Botswana, Burkina Faso, Burundi, Cameroon, Cape Verde, Central African Republic, Chad, Comoros, Congo, Cote d'Ivoire, Egypt, Equatorial Guinea, Eritrea, Ethiopia, Gabon, Gambia, Ghana, Guinea, Kenya, Liberia, Libya, Madagascar, Malawi, Mali, Mauritania, Mauritius, Morocco, Mozambique, Namibia, Nigeria, Reunion, Rwanda, Senegal, Siera Leone, Somalia, South Africa, Sudan, Swaziland, Tanzania, Togo, Tunisia, Uganda, Zaire, Zambia, and Zimbabwe. **Asia:** Afghanistan, Bangladesh, Brunei, and India. **Europe:** France, Germany, Greece, Italy, Malta, Portugal, Spain, and United Kingdom. **Middle East:** Bahrein, Cyprus, Iran, Iraq, Israel, Jordan, Lebanon, Oman, Saudi Arabia, Syria, United Arab Emirates, and Yemen. **Oceania:** American Samo

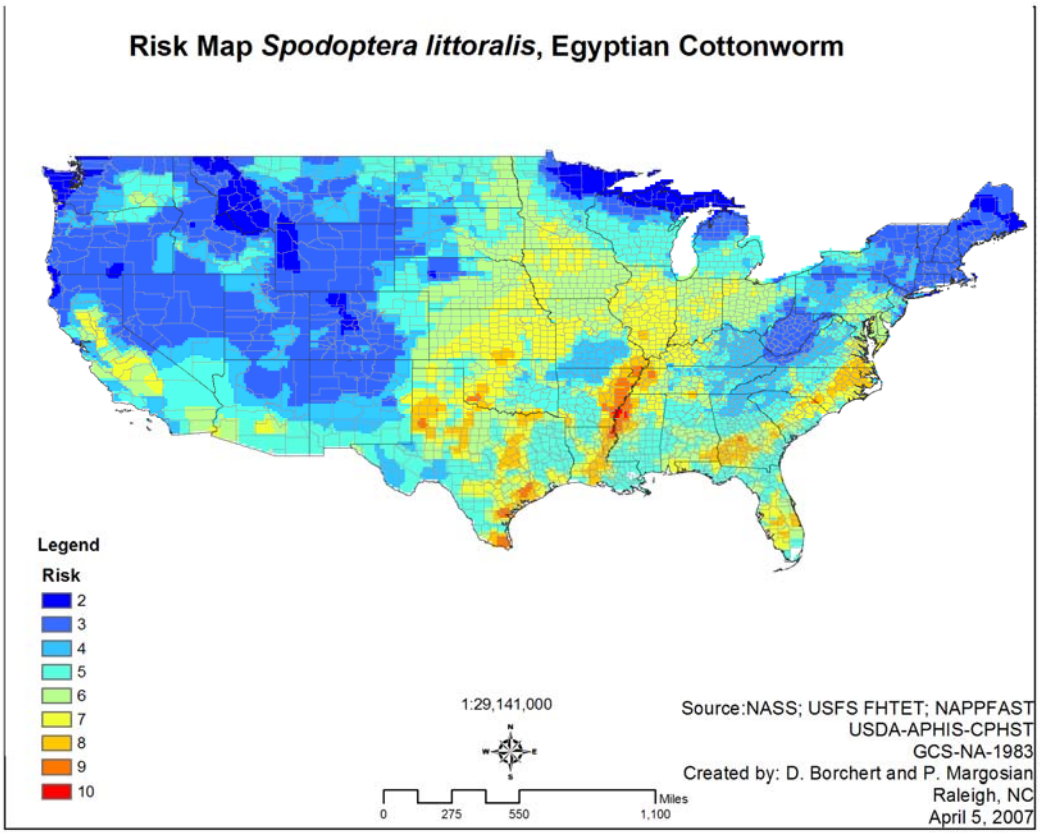

Figure 4. Risk map for *S. littoralis* within the continental United States. The greater the risk (climate and host availability), the greater the risk number.Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Potential Distribution Within the United States

The pest has been intercepted at U.S. ports on plant parts, leaves, and flowers. The potential U.S. range of most *S. littoralis* may be limited to the west coast through the lower southwestern and southeastern United States, reaching as far north as Maryland (USDA, 1982). Migratory species may be capable of periodic spread into northern states and even Canada by late summer or early fall.
A recent risk map developed by USDA-APHIS- PPQ-CPHST (Fig. 4) shows that portions of Arkansas, Lousiana, and Mississippi are at the greatest risk from *S. littoralis*. In addition, portions of Kansas, Missouri, Illinois, Oklahoma, Texas, Tennessee, Alabama, Georgia, Florida, North Carolina, South Carolina, and Virginia have a risk level of 8 or higher.

Survey (From Venette et al., 2003; CABI, 2004)

Preferred Method: Pheromone traps can be used to monitor the incidence of *S. littoralis* (Rizk et al., 1990). The synthetic sex pheromone (*Z*,*E*)-(9,11)-tetradecadienyl acetate has proven highly effective at trapping male moths of *S. littoralis* (Salem and Salama, 1985). Kehat and Dunkelblum (1993) found that the minor sex pheromone component, (9Z,12Z)-9,12-tetradecadienyl acetate in addition to the major component (9Z,11Z)-9,11-tetradecadienyl acetate was required to attract males.

A lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory. The lure (a 200:1 mixture of (Z, E)-(9-11)-tetradecadieyl acetate to (Z,E)-9,12)-tetradecadienyl acetate is formulated in a Beem capsule with a 2-week field life. For large orders, laminates are formulated with a 12-week field life.

Sex-pheromone baited delta traps remained attractive for approximately 2 weeks, but effectiveness declined after 3 to 4 weeks of use (Ahmad, 1988). To monitor male flight activity in vegetable production areas, delta traps were placed 1.7 m above the ground at a rate of 2 traps/ha (approximately 1 trap/acre) (Ahmad, 1988). Pheromone lures impregnated with 2 mg of the pheromone blend (blend not specified) were replaced after 4 weeks of use (Ahmad, 1988). Traps are deployed at a similar height (1.5 m) to monitor male flight in cotton (Salem and Salama, 1985).

Lures for *S. littoralis* can be used in the same traps with lures for *S. litura*, *Helicoverpa armigera*, *Pectinophora scutigera* (all not known to occur in the US), and *P. gossypiella* (exotic established in US). Lures for *S. littoralis* may also attract *Erastria* sp. (established in US) (PPQ, 1993).

Alternative Method: Visual surveys for this pest can take place any time during the growing season while plants are actively growing (usually spring through fall in temperate areas). Early instars (<3rd) are likely to be on lower leaf surfaces during the day. The larvae will skeletonize leaves by feeding on this surface and

such damage to the leaf provides evidence of the presence of larvae. Sweep net sampling may be effective at dawn or dusk.

Specimen identification should be confirmed by a trained taxonomist (USDA, 1982). However, not all sampling methods are equally effective for all life-stages of the insect. Eggs are only likely to be found by visual inspection of leaves. First through third instars may be detected by sweep net sampling; nearly all instars can be detected by visual inspection of plants; and, later instars (4th-6th) and pupae may be found by sieving soil samples (Abul-Nasr and Naguib, 1969; Abul-Nasr et al.,1971).

Not recommended:
Light traps using a 125 W mercury-vapor bulb have been used to nondiscriminately capture multiple *Spodoptera* spp. (Blair, 1974) and most assuredly other insects as well. A modified light trap using six 20-W fluorescent lights also proved effective for monitoring flight activity of *S. littoralis* (El-Mezayyen et al., 1997). However, light traps can attract many types of insects, including similar-looking Lepidopteran species, and are therefore not recommended for use in early detection surveys.

For additional survey information see:
http://www.aphis.usda.gov/import_export/plants/manuals/emergency/downloads/nprg_spodoptera.pdf

Key Diagnostics

Close observation of adult genitalia is often the only certain method to separate species.

Easily Confused Pests

S. littoralis is often confused with *S. litura* and the variability and similarity of the two species makes correct identification difficult; examination of adult genitalia is often the only certain method to separate the two species. For more information on morphological discrimination between the adult, pupal and larval stages of the two species, refer to Schmutterer (1969), Cayrol (1972), Mochida (1973) and Brown and Dewhurst (1975).

Although markings on larvae are variable, a bright-yellow stripe along the length of the dorsal surface is characteristic of *S. litura*. On dissection of the genitalia, the ductus and ostium bursae are the same length in female *S. littoralis*, whereas they are different lengths in *S. litura*. The shape of the juxta in males in both species is very characteristic,

Figure 5. Larva of *S. exigua*. Photo courtesy of Oklahoma State University.

and the ornamentation of the aedeagus vesica is also diagnostic. The genitalia must be removed, cleaned in alkali and examined microscopically. *S. litura* is not established in the continental US, but has been reported in Hawaii.

Larvae of *S. littoralis* can be confused with *S. exigua*, the beet armyworm (established in the United States) (Fig. 5), but *S. littoralis* larvae are light or dark brown, while *S. exigua* are brown or green. *S. littoralis* is also larger than *S. exigua* (Venette et al., 2003).

Adults of *S. littoralis* are almost nearly identical in appearance to *S. ornithogalli*, the yellow striped armyworm (Fig. 6), a common pest in the United States. The hind wings of female *S. littoralis* are darker than those of *S. ornithogalli* (USDA, 1982).

Figure 6. Adult of *S. ornithogalli*. Photo courtesy of Mississippi Entomological Museum. http://mothphotographersgroup.msstate.edu/Files/JV/JV50.7.shtml

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Secondary Pests of Small Grains (Truncated Pest Datasheet)

Heteronychus arator

Scientific Name
Heteronychus arator (Fabricius)

Synonyms:
Heteronychus sanctaehelenae, Heteronychus transvaalensis, Scarabaeus arator

Common Names
African black beetle, black maize beetle, black lawn beetle, black beetle

Type of Pest
Beetle

Taxonomic Position
Class: Insecta, **Order**: Coleoptera, **Family**: Scarabaeidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description
Life stages are shown in Figures 1 and 2.

Eggs: White, oval, and measuring approximately 1.8 mm long at time of oviposition. Eggs grow larger through development, and become more round in shape. Eggs are laid singly at a soil depth of 1 to 5 cm. Females each lay between 12 to 20 eggs total. In the field, eggs hatch after approximately 20 days. Larvae can be seen clearly with the naked eye (CABI, 2004; Matthiessen and Learmoth, 2005).

Larvae: There are three larval instars. Larvae are creamy-white except for the brown head capsule and hind segments, which appear dark where the contents of the gut show through the body wall. The head capsule is smooth textured, measuring 1.5 mm, 2.4 mm, and 4.0 mm at each respective instar. The

Figure 1. Illustration of each stage of the life cycle of the African black beetle, showing a close up view of each stage and a background view showing that the eggs, larvae, and pupae are all underground stages with the adult beetles as the only stage appearing above ground. Illustration courtesy of NSW Agriculture. http://www.ricecrc.org/Hort/ascu/zecl/zeck113.htm

third-instar larva is approximately 25 mm long when fully developed. African black beetle larvae are soil-dwelling and resemble white 'curl grubs.' They have three pairs of legs on the thorax, a prominent brown head with black jaws, and are up to 25 mm long. The abdomen is swollen, baggy, and grey/blue-green due to the food and soil they have eaten. Larvae eat plant roots, potentially causing significant damage to turf, horticultural crops, and ornamentals. Turf is the preferred host of the larvae (CABI, 2004; Matthiessen and Learmoth, 2005).

Figure 2. Eggs, larvae, and adult African black beetle. Photo courtesy of Yates Ltd.
http://www.yates.com/au/ProblemSolver/BlackBeetle.asp

Pupae: The larvae, when fully grown, enter a short-lived pupal stage, which measures approximately 15 mm long and is typically coleopteran in form (cylindrical shape), initially pale yellow, but becoming reddish-brown nearer to the time of emergence (Matthiessen and Learmoth, 2005).

Adults: Beetles are 12 to 15 mm long; shiny black dorsally and reddish-brown ventrally. The females are slightly larger than males. Males and females are readily differentiated by the shape of the foreleg tarsus. The tarsus of the male is much thicker, shorter, and somewhat hooked compared with that of the female, which is longer and filamentous. A less obvious sexual difference is in the form of the pygidium at the end of the abdomen. In the male, it is broadly rounded ,and in the female, it is apically pointed. The beetle is the main pest stage (CABI, 2004; Matthiessen and Learmoth, 2005).

Symptoms/Signs

The adult is the main pest stage. The adult is the only aboveground stage and is capable of flight. The beetles are of considerable economic importance because they attack a wide range of plants. The beetle damages pastures, particularly newly-sown ryegrass and perennial grasses, millet, corn, turf, barley, triticale, wheat crops (not oats), a wide range of vegetable crops, grape vines, ornamental plants and newly-planted trees. Larvae damage turf and underground crops, notably potato tubers (Matthiessen and Learmoth, 2005).

Stems experience external feeding, and the whole plant may be toppled or uprooted. Adult damage to plants typically involves chewing of the cortex of stems just below the surface of the ground. High densities of *H. arator* in pastures lead to clover (a non-host) becoming dominant over grasses (Matthiessen and Learmoth, 2005).

Survey

Preferred Method: Visual survey is the preferred method to survey for *H. arator.* Areas that are rotated with or replace pasture lands are most at risk of damage from the African black beetle. Most damage by the African black beetle occurs during the spring to early summer when the adults are most active crawling on the soil surface and again after new adults emerge in mid summer to fall. Inspect immediately below the soil surface for signs of *H. arator* attack, in particular frayed chewing around the stem circumference.

In grass and turf, heavy infestations can be detected by lifting up tufts of grass and inspecting for abundant frass or distinct channeling of soil with embedded larvae. Less dense infestations will be evident if sections of grass are dug and examined for presence of larvae or adults.

Mathiessen and Learmonth (1993) devised a method for sampling *H. arator* in potato crops where the pest was known to be present. A modified version of their approach may be useful for surveys in other corps. In their survey, 50cm long portions of hilled-up rows comprised the sample unit. A 70 x 30 cm piece of sheet steel is pressed into soil across the row with soil on one side of the metal sheet being excavated. The steel sheet is then removed to expose an undisturbed soil face of the potato hill. Presence of the beetle or other pests and associated plant damage in the top, center, or bottom of soil cross sections is recorded. Fifty samples were examined at each sampling time in a uniform grid across a 0.2 ha crop area.

Alternative Methods: Matthiessen and Learmonth (1998) used pitfall, light, and window traps to monitor *H. arator* in Australia. Light traps are often used in Australia to monitor adult flight activity during the summer and fall prior to planting on old pasture or potato land. Light traps were similar to a Pennsylvania trap (Southwood, 1978). The light was a vertically-oriented 60 cm-long 20 watt fluorescent black light, the center of which was 1.5 meters above the ground.

Four vertically-oriented 17.5-cm wide panels equi-radial from the light served as baffles to arrest the flight of insects attracted to the light, causing them to fall through a 21 cm diameter funnel into a collecting container holding an insecticidal vapor strip. A timer kept the light on daily from sunset to sunrise. Light traps were cleared weekly.

Because the beetles are clumsy walkers, they can be collected by pitfall traps or sharp sided plough lines. Matthiessen and Learmonth (1998) made pitfall traps from a 21 cm diameter funnel fitted at ground level into a buried PVC cylinder. Insects fell into a 21 cm plastic jar containing 500 ml of 1:1 ethylene glycol and water. Mesh panels on the upper sides of the collecting jar allowed rainfall to drain away. These traps were spaced at 10 meter intervals at one location. Subsequent traps were made from a 10 cm diameter plastic funnel glued into the screw-top lid of a 250 ml plastic jar. These smaller traps were more easily placed in pasture by creating a hold with a 10 cm diameter corer, and inserting the whole trap assembly. No preservative was used for these smaller traps due to the small size and absence of large predators capable of consuming adult *H. arator*. The narrow neck of the funnel fastened into the lid prevented escape of beetles, and holes at the base of the collecting jar allowed drainage. Typically, ten traps were deployed, at 5 meter intervals in each of two lines 10 meters apart. Captures in all pitfall traps were assessed weekly.

Soil sampling is also used to monitor populations of *H. arator* in Australia. Adults were counted from shovels full of soil, and six beetles per square meter represented a potentially damaging population (Matthiessen and Learmonth, 1998). To estimate the density of *H. arator* in pastures 100 soil core samples, 10 cm in diameter x 15 cm deep were used. The soil cores were broken up at the time they were taken and searched only for easily seen large life stages of *H. arator* that occur in the summer and autumn (Matthiessen and Ridsdill-Smith, 1991).

Key Diagnostics

African beetle larvae can be identified with the naked eye, since their anal opening is horizontal, compared with a vertical opening in other species. Smith et al. (1995) provided detailed illustrated descriptions and a laboratory and field key to third star larvae. Cumpston (1940) also described the features of the larvae that allow *H. arator* to be distinguished from other species. Keys to identify adults from related species are given by Enrodi (1985).

Easily Confused Pests

The larvae can be confused with lesser pasture cockchafer (*Australaphodius frenchi*) in Australia. However, larvae of the lesser pasture beetle are never larger than first instar African black beetle and are much shorter (only up to 3 to 4 mm). To avoid confusion between *H. arator* and native cockchafers, close examination is necessary (Matthiessen and Learmoth, 2005). After reviewing the

literature, it appears that *Australaphodius frenchi* is not currently present in the United States.

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Lobesia botrana

Scientific Name
Lobesia botrana Denis & Schiffermüller

Synonyms:
Cochylis vitisana, Cochylis botrana, Coccyx botrana, Eudemis botrana, Eudemis rosmarinana, Grapholita botrana, Lobesia rosmariana, Noctua romani, Paralobesia botrana, Penthina vitivorana, Polychrosis botrana, Tortrix botrana, Tortrix vitisana, Tinea premixtana, Tinea reliquana, *Tortrix reliquana, Tortrix romaniana*

Common Names
European grape vine moth, grape berry moth, grape fruit moth, grape leaf-roller, grape vine moth, grape moth, vine moth

Type of Pest
Moth

Taxonomic Position
Class: Insecta **Order**: Lepidoptera **Family**: Tortricidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description **(from CABI, 2004)**
Eggs: The egg of *L. botrana* is of the so-called “flat type”, with the long axis horizontal and the micropile at one end. Elliptical, with a mean eccentricity of 0.65, the egg measures about 0.65 to 0.90 x 0.45 to 0.75 mm. Freshly laid eggs are pale cream or yellow, later becoming light gray and translucent with iridescent glints. The chorion is macroscopically smooth but presents a slight polygonal reticulation in the border and around the micropile. As typically occurs in the subfamily Olethreutinae, eggs are laid singly, and more rarely in small clusters of two or three.

Larvae: There are usually five larval (Fig. 1) instars. Neonate larvae are about 0.95 to 1 mm long, with head and prothoracic shield deep brown, nearly black, and body light yellow. Mature larvae reach a length between 10 and 15 mm, with the head and prothoracic shield lighter than neonate larvae and the body color

varying from light green to light brown, depending principally on larval nourishment.

Pupae: Female pupae are larger (5 to 9 mm) than males (4 to 7 mm). Freshly formed pupae are usually cream or light brown but also light green or blue, and a few hours later become brown or deep brown (Fig. 2).

The sexes may be distinguished by the position of genital sketches that are placed in the IX and VIII abdominal sternites in males and females, respectively. Moreover, the male genital orifice is placed between two small lateral prominences. When adult emergence is imminent, pupae perforate the cocoon, resting the exuvia fixed outwardly in a characteristic position by cremaster spines.

Adults: Adults are 6 to 8 mm long with a wingspan of about 10 to 13 mm. The head and abdomen are cream colored; the thorax is also cream with black markings and a brown ferruginous dorsal crest. The legs have alternate pale cream and brown bands.

Forewings have a mosaic-shaped pattern with black, brown, cream, red and blue ornamentation (Fig. 3). The ground color is bluish gray and fasciae brown, shaped by a pale cream border; scales lining the costa, termen and dorsum are darker than the wing ground color.

Cilia are brown with a paler apical tip and a cream basal line along the termen.

The underside is brownish gray, gradually darker towards the costa and apex. Cilia and cubital tuft are grayish brown with a paler basal line.

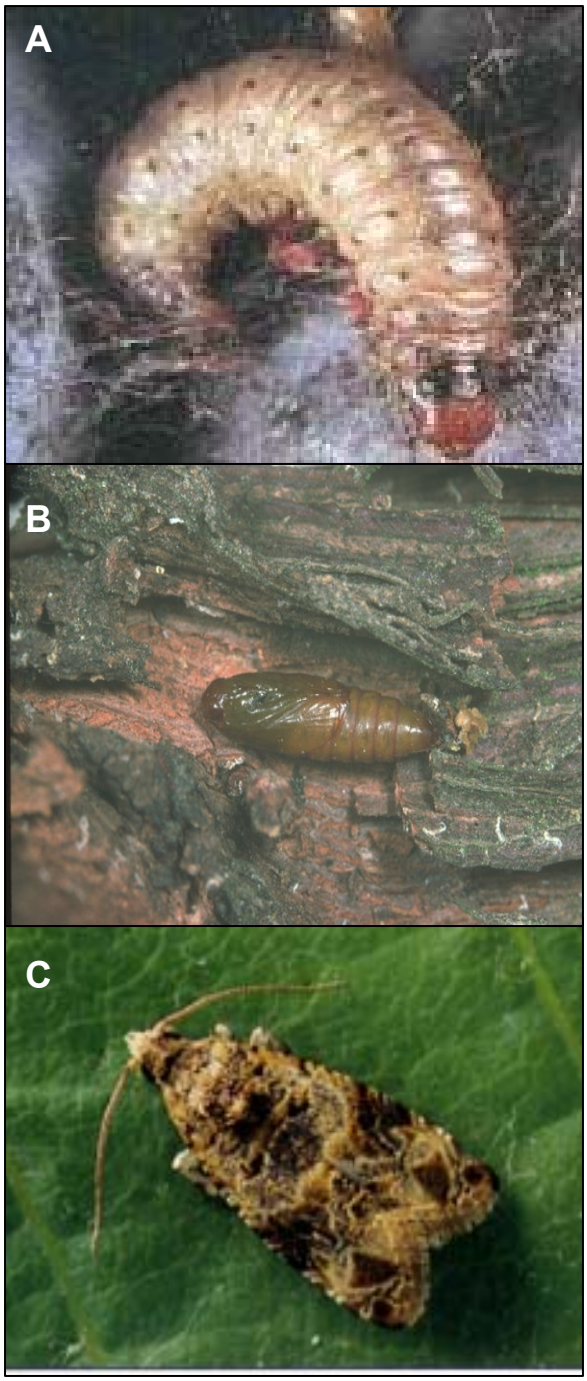

Figure 1. Larva (A), pupa (B), and adult (C) *L. botrana.* Photos courtesy of Instituto Agrario S. Michele All' Adigen, HYPPZ Zoology, and pherobase.net, respectively

There is no clear sexual dimorphism, but the sexes may be easily separated by their general morphology and behavior: as in the pupal stage, males are smaller than females, they have a narrower abdomen with an anal fine comb of modified scales (hair pencils), and when disturbed they exhibit movements more quick and nervous than those of females.

Symptoms/Signs

On inflorescences (first generation), neonate larvae firstly penetrate single flower buds. Symptoms are not evident initially, because larvae remain protected by the top bud. Later, when larval size increases, each larva agglomerates several flower buds with silk threads forming **glomerules** visible to the naked eye (Fig. 2), and the larvae continue feeding while protected inside. Larvae usually make one to three glomerules during their development. Despite hygienic behavior of larvae, frass may remain adhering to the glomerules (CABI, 2004).

Figure 2. Glomerules of *L. botrana.* Photo courtesy of EFAPO-ES. http://www.efa-dip.com

First-generation larval feeding on the buds or flowers webs them and prevents further growth. If heavy flower damage occurs during the first moth generation, the affected flowers will fail to develop and yield will be low. Damage by summer larvae of the second and third generations result in many nibbled berries, which later shrivel. The berries may be eaten either partly (leading to rot) or completely (leaving only empty skins at the tip of the bunch). Sometimes berries drop, and only the stalks remain (USDA, 1985).

Survey

From Venette et al. (2003)

Preferred Method: A sex pheromone has been identified that is highly attractive to males. Males are most attracted to a five component blend of (*E*,*Z*)-(7,9)-dodecadienyl acetate, (*E*,*Z*)-(7,9)-dodecadien-1-ol, (*Z*)-9-dodecenyl acetate, (*E*)-9-dodecenyl acetate, and 11-dodecenyl acetate in a ratio of 10:0.5:0.1:0.1:1. Males are slightly less attracted to a three component blend of (*E*,*Z*)-(7,9)-dodecadienyl acetate, (*E*,*Z*)-(7,9)-dodecadien-1-ol, (*Z*)-9-dodecenyl acetate (ratio of 10:0.5:0.1). Males were still attracted, but much less so, to the main pheromone component (*E*,*Z*)-(7,9)-dodecadienyl acetate. The main pheromone

component has been used to disrupt mating as a method of pest control and to monitor the flight period of males. However, this compound is sensitive to sunlight and degrades, becoming non-attractive to *L. botrana*, after 60 minutes of exposure to UV radiation. **A pheromone lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory. The lures is loaded with 0.5 mg of (*E*,*Z*)-(7,9)-dodecadienyl acetate (see precautions above).**

Pheromone-baited traps (e.g., Pherocon 1C, Zoecon) have been used to monitor male flight activity (Anshelevich et al., 1994) and to make informed treatment decisions in grape production areas. Traps placed 4 ft high (1.3 m) are generally more effective than traps placed at only 1 ft (0.3 m). Delta traps catch relatively fewer moths than traps with a more open design (e.g., Traptest traps described as "commercial type (Montedison, Milan, Italy), consisting of two triangular plastic roofs in Havana brown; sticky area 9.89 dm^2 [152 in^2]"). When pheromone traps are used, care should be taken to keep foliage away from the entry to the trap (PPQ, 1993). Rubber septa used to dispense the pheromone should be replaced every 3 weeks (PPQ, 1993). Traps should be placed approximately 100 ft (30.5 m) apart to avoid inter-trap interference. Lures for *L. botrana* can be used in the same trap with lures for *Lymantria dispar* or *Cydia pomonella* (Schwalbe and Mastro, 1988).

Alternative Method: USDA (1985) suggests visually inspecting for eggs on flower buds or pedicels of vines and grapes. It is preferable to look for larval damage rather than for eggs, because detection of eggs is very tedious and time-consuming, especially under field conditions. Look for webbed bud clusters (glomerules) or flowers where the spring generation larvae feed. Inspect for pupae under rolled leaves in spring. Inspect grapes and look for eggs or damaged berries. Cut open grapes and search for summer generation larvae and pupae.

Suspect adult specimens should be pinned and labeled for subsequent identification. Submit suspect larvae or pupae in alcohol. For field surveys, Badenhauser et al. (1999) recommended a sample unit of a grape vine. Sample units should be selected at random.

Not recommended: Light traps have been used but their lack of specificity makes their use inadvisable when the appropriate pheromones are available. Feeding traps were largely used in the past before pheromone traps were developed, but may still be useful in particular situations. An earthen or glass pot is baited with a fermenting liquid (fruit juice, molasses, etc.) and the scents produced attract adults, which are then drowned. Practical problems include irregularity in trapping because fermentation strongly depends on seasonal temperature, trap maintenance (lure replenishment and foam elimination), and low selectivity.

A corrugated paper band technique has sometimes been employed to trap and quantify overwintering pupae. Bands are placed around grapevine trunks or primary branches, and diapausing larvae pupate inside. However, this method is only useful in the last generation, and its reliability is uncertain.

Key Diagnostics

Hindwing coloration and the male clasper lacks spine at base (Vennette et al. 2003).

Easily Confused Pests

In the Palaearctic vine-growing areas, other lepidopteran species have an ecological niche similar to that of *L. botrana*, including *Eupoecilia ambiguella*, *Argyrotaenia pulchellana* [*Argyrotaenia ljungiana*], *Clepsis spectrana*, *Cryptoblabes gnidiella*, *Euzophera bigella* and *Ephestia parasitella*. However, only the first of these, *E. ambiguella* (Fig. 3), may cause comparable damage to *L. botrana*, at least in northern European vineyards. Adults of these species may be easily differentiated macroscopically using a photographic key. *E. ambiguella* forewings are cream with a median fascia bluish dark brown. In field conditions, larvae may be distinguished because (i) the head of *E. ambiguella* is darker than that of *L. botrana*; (ii) *L. botrana* larvae do not carry any protective silk cover; and (iii) the behavior of *L. botrana* when disturbed is quicker and even violent. Moreover, *L. botrana* pupation occurs inside a grayish white cocoon that usually does not incorporate vegetal residues and frass, as occurs in *E. ambiguella* (CABI, 2004).

Figure 3. *Eupoecilia ambiguella* adult and pupa.
Photos courtesy of HYPP Zoology.

Another tortricid species, the American grape berry moth, *Endopiza viteana* (Fig. 4) [*Polychrosis viteana*], occurs in the eastern USA, and presents similar bionomics to *L. botrana* (Roehrich and Boller, 1991).

Figure 4. American grape berry moth, *Endopiza viteana*. Photo courtesy of Michigan State University.

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Spodoptera litura

Scientific Name
Spodoptera litura Fabricius

Synonyms:
Mamestra albisparsa, Noctua elata, Noctua histrionica, Noctua litura, Prodenia ciligera, Prodenia declinata, Prodenia evanescens, Prodenia glaucistriga, Prodenia litura, Prodenia subterminalis, *Prodenia tasmanica, Prodenia testaceoides*, *Prodenia littoralis, Spodoptera littoralis*

Common Name(s)
Rice cutworm, armyworm, taro caterpillar, tobacco budworm, cotton leafworm, cluster caterpillar, cotton worm, Egyptian cotton leafworm, tobacco caterpillar, tobacco cutworm, tobacco leaf caterpillar, common cutworm

Type of Pest
Moth

Taxonomic Position
Class: Insecta, **Order:** Lepidoptera, **Family:** Noctuidae

Reason for Inclusion in Manual
CAPS Target (2009): AHP Prioritized Pest List

Pest Description
The two Old World cotton leafworm species, *Spodoptera litura* and *S. littoralis*, are allopatric, their ranges covering Asia and Africa, Europe and the Middle East, respectively. Many authors have regarded them as the same species, but they have been differentiated based on adult genitalia differences (Mochida, 1973; CABI, 2004).

Eggs: Spherical, somewhat flattened, sculpted with approximately 40 longitudinal ribs, 0.4 - 0.7 mm in diameter; pearly green, turning black with time, laid in batches covered with pale orange-brown or pink hair-like scales from the females body (Pearson, 1958; CABI, 2004).

Larva: Newly hatched larvae are tiny, blackish green with a distinct black band on the first abdominal segment. Fully grown larvae are stout and smooth with scattered short setae. Head shiny black, and conspicuous black tubercules each with a long hair on each segment. Color of fully grown larvae not constant, but varies from dark gray to dark brown, or black, sometimes marked with yellow dorsal and lateral stripes of unequal width. The lateral yellow stripe bordered dorsally with series of semilunar black marks. Mature larvae are 40-50 mm. Two

large black spots on first and eight abdominal segments (Hill, 1975; USDA, 1982; CABI, 2004).

Figure 1. Egg mass (left), larva (center), and adult (right). Photos courtesy of CABI, 2004.

Pupa: Reddish brown in color, enclosed inside rough earthen cases in the soil, 18-22 mm long, last abdominal segment terminates in two hooks (USDA, 1982; CABI, 2004).

Adult: Body whitish to yellowish, suffused with pale red. Forewings dark brown with lighter shaded lines and stripes. Hind wings whitish with violet sheen, margin dark brown and venation brown. Thorax and abdomen orange to light brown with hair-like tufts on dorsal surface. Head clothed with tufts of light and dark brown scales. Body length 14-18 mm, wing span 28-38 mm (Hill, 1975; USDA, 1982).

See Schmutterer (1969), Cayrol (1972), and Brown and Dewhurst (1975) for additional information.

Symptoms/Signs

Specific information on wheat is not available. On most crops, damage arises from extensive feeding by larvae, leading to complete stripping of the plants. Larvae are leaf eaters but sometimes act as a cutworm with crop seedlings.

Spodoptera litura feeds on the underside of leaves causing feeding scars and skeletonization of leaves. Early larval stages remain together radiating out from the egg mass. However, later stages are solitary. Initially there are numerous small feeding points, which eventually spread over the entire leaf. Because of this pest's feeding activities, holes and bare sections are later found on leaves, young stalks, bolls, and buds. Larvae mine into young shoots. In certain cases, whole shoot tips wilt above a hole and eventually die (Hill, 1975; USDA, 1982).

On cotton, leaves are heavily attacked and bolls have large holes in them from which yellowish-green to dark-green larval excrement protrudes. In tobacco, leaves develop irregular, brownish-red patches and the stem base may be gnawed off. The stems of corn are often mined and young grains in the ear may be injured (CABI, 2004).

Survey

Preferred Method: The identification of a male sex pheromone of *S. litura*, (Z,E)-(9,11)-tetradecadienyl acetate and (Z,E)-(9,12)-tetradecadienyl acetate by Tamaki (1973) has enabled effective monitoring of this species for several years. One milligram of a 10:1 mixture of these two compounds in a rubber septum attracted a comparable number of males as 10 caged virgin females in the field (Yushima et al., 1974). The compounds are most effective in a ratio (A:B) between 4:1 to 39:1 (Yushima et al., 1974). The two components in a ratio of 9:1 are available commercially as Litlure in Japan (Yushima et al., 1974). For early detection sampling, traps should be placed in open areas with short vegetation (Hirano, 1976; Venette et al., 2003).

Trap height: Krishnananda and Satyanarayana (1985) found that trap catches at 2.0 m above the ground level caught significantly more male *S. litura* than those placed at higher of lower heights (ranging from 0.5 m to 4.0 m). Ranga Rao et al. (1991) suggest trap placement at 1 m.

Alternative Method: Visual survey can be used to determine the presence of *S. litura*. The presence of newly hatched larvae can be detected by the 'scratch' marks they make on the leaf surface. Particular attention should be given to leaves in the upper and middle portion of the plants (Parasuraman, 1983). The older larvae are night-feeders, feeding primarily between midnight and 3:00 am and are usually found in the soil around the base of plants during the day. They chew large areas of the leaf, and can, at high population densities, strip a crop of its leaves. In such cases, larvae migrate in large groups from one field to another in search of food. *S. litura* may be detected any time the hosts are in an actively growing stage with foliage available, usually spring and fall. Check for 1st and 2nd instar larvae during the day on the undersurface of leaves and host plants. Watch for skeletonized foliage and perforated leaves. If no larvae are obvious, look in nearby hiding places. Third instar larvae rest in upper soil layers during the day. Sweep net for adults and larvae at dawn or dusk. Watch for external feeding damage to fruits. Watch near lights and light trap collections for adult specimens. Submit similar noctuid moths in any stage for identification (USDA, 1982).

Light traps have been used to monitor *S. litura* populations (Vaishampayan and Verma, 1983). Capture of *S. litura* moths was affected by the stage of the moon, with the traps being least effective during the full moon and most effective during the new moon (Parasuraman and Jayaraj, 1982).

A recent risk map developed by USDA-APHIS- PPQ-CPHST (Fig. 2) shows that portions of Arkansas, Florida, Georgia, Lousiana, Mississippi, and Texas are at the greatest risk from *S. littoralis*. In addition, portions of Alabama, Arizona, California, Missouri, Oklahoma, North Carolina, South Carolina, and Tennessee have a risk level of 8 or higher. The pest has been present in Hawaii since 1964 (CABI, 2004). *S. litura* was identified in a sample from a Miami-Dade County,

Florida nursery in April 2007. Pheromone traps have been placed over a nine square mile areas and have yielded no additional finds.

Key Diagnostics

Wing coloration has been used to separate the sexes of *S. litura* (Singh et al., 1975). *Spodoptera litura* can be easily confused with *S. littoralis* as in both cases adults are similar, and they can be distinguished only through examination of genitalia. On dissection of the genitalia, ductus and ostium bursae are the same length in female *S. littoralis,* different lengths in *S. litura*. The shape of the juxta in males is very characteristic, and the ornamentation of the aedeagus vesica is also diagnostic. The larvae of the two species are not easily separable, but some distinguishing criteria are used for the 6th instar. Mochida (1973) provides information on morphological discrimination between the adult, pupal and larval stages of the two species.

For additional images, including photos of host damage see http://www.padil.gov.au/viewPestDiagnosticImages.aspx?id=418

Easily Confused Pests

Adult *S. litura* closely resemble *Spodoptera ornithogali* (yellowstriped armyworm), a pest in the United States. However, the hindwings of female *S. litura* are darker than those of *S. ornithogalli*.

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Tertiary Pests of Small Grains (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to small grains and exotic to the US

None at this time

Mollusks

Primary Pests of Small Grains (Full Pest Datasheet)

None at this time

Secondary Pests of Small Grains (Truncated Pest Datasheet)

Cernuella virgata

Scientific name
Cernuella virgata Da Costa

Synonyms:
Cernuella virgatus, *Cernuella variabilis, Cernuella virgata* ssp. *variegate*, *Helicella maritime*, *Helicella variabilis*, *Helicella virgata*, *Helix virgata*

Common Name(s)
Maritime gardensnail, Mediterranean snail, Mediterranean white snail, striped snail, vineyard snail, white snail

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Stylommatophora (Eupulmonata), **Family:** Hygromiidae (Helicidae)

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description

The maritime garden snail is relatively small and is characterized by prominent spiral banding on the shell (Fig. 1). The shell of *C. virgata* is globose-depressed and white or yellowish-white in color with dark-brown bands or spots (Fig. 1, 2). Snail size is 6 to 19 mm high x 8 to 25 mm wide. Shell size and banding patterns are reported to vary widely geographically throughout Southeastern Australia (Baker, 1988). Size has been demonstrated as inversely proportional to population density (Baker, 1988). *C. virgata* is considered polymorphic; banded and unbanded (more common) morphs have been found throughout Australia. Relative frequencies of each morph are likely correlated with site-specific factors such as predator pressure (Baker, 1988).

Figure 1. Banding of *C. virgata*. Photo courtesy of Tenby Museum

Figure 2. *C. virgata*. Photo courtesy of L. Poggiani. www.lavalledelmetauro.it

Note: *Cernuella virgata* has been introduced in the states of Washington (NAPIS, 2007) and California (Hardy, 2004).

Symptoms/Signs

Cernuella virgata is found atop plants during summertime (Fig. 3) and may also be found feeding on new growth earlier in the season. These snails aestivate on plant heads and stalks, which contaminate crops and clog machinery. Areas previously infested with snails can prevent re-establishment of site as pastureland, as livestock often reject slime-contaminated hay and forage (Baker, 2002).

Survey

Preferred Method: The most effective method of survey for mollusks is through visual survey methods. While conducting a survey, look for snail eggs, juveniles and adults, as well as clues that suggest the presence of mollusk pests which may include: empty snail shells, mucus and “slime” trails, and/or ribbon-like feces.

Cernuella virgata is a conspicuous crop pest that hides during the day. This pest often has a patchy distribution within a field. Surveys are best carried out at night using a flashlight or in the morning or evenings following a rain event. It is easily seen, and attacked plants exhibit extensive rasping and defoliation.

At this time, a host risk map is available (Fig. 3). Surveys should take place at areas of greatest risk. The host risk map describes the relative density (on a scale of 1-10) of susceptible hosts. The maps are based on National Agricultural Statistics Service (NASS) data. The scale of one to ten describes the proportion of total host acreage per county: for example a rank of one indicates no host acreage, while a score of ten indicates that 75-100% of the acres in the county contain suitable hosts for the pest. This map shows that portions of Arkansas, Indiana, Illinois, Kansas, Kentucky, Missouri, Ohio, and Tennessee are at the greatest risk (rating of 5) from this mollusk based on host availability. Portions of Colorado, Delaware, Idaho, Iowa, Lousiana, Maryland, Michigan, Minnesota, Mississippi, Montana, New Jersey, New York, North Carolina, Oklahoma, Oregon, Pennsylvania, South Carolina, Texas, Virginia, Washington, and Wisconsin are also at risk based on host availability (rating of 4).

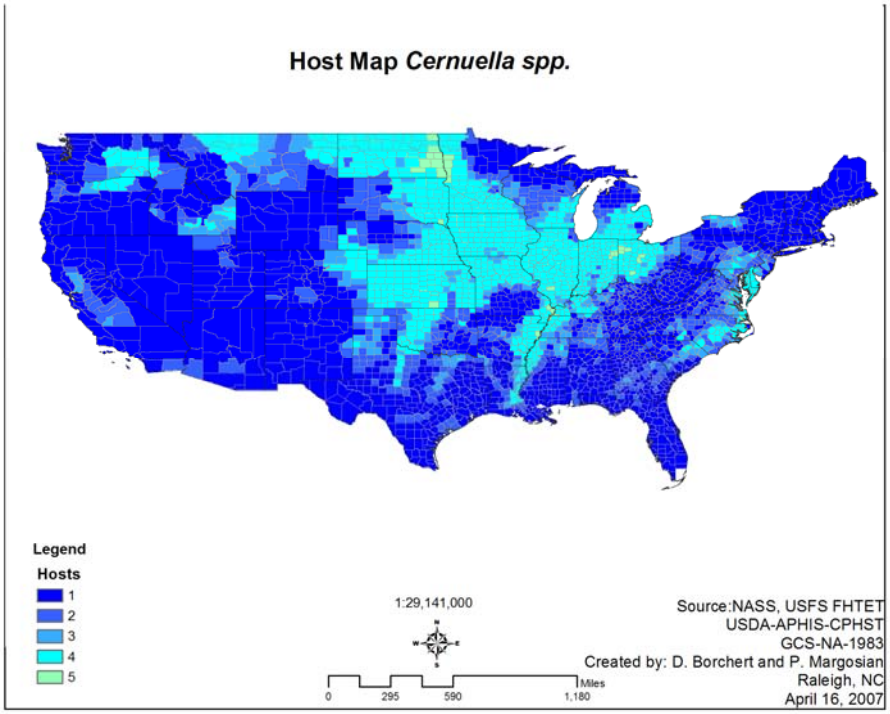

Figure 3. Host risk map for *Cernuella spp.* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Note: Serious diseases are associated with the consumption and improper handling of certain mollusks (snails and slugs). Of particular concern,

many mollusk species serve as intermediate hosts of nematodes and trematodes. While most cases of human infections result from consumption of raw or partially cooked snail meat, government inspectors, officers and field surveyors are at-risk due to the handling of live snail, samples, and potential exposure to mucus secretions. ***Wear neoprene gloves when handling mollusks and wash hands thoroughly after any mollusk survey or inspection activities.***

Key Diagnostics

Cernuella virgata is a relatively small snail (up to 15mm in diameter) characterized by prominent spiral banding on the shell.

Closely resembles the white Italian snail (*Theba pisana*) in appearance and pest status. *Cernuella virgata* can be differentiated from *T. pisana* based on more pronounced spiral banding and the umbilicus (hole about which the shell spirals) appears as a circular hole rather than being partially obscured as in the white Italian snail (CABI, 2004).

Figure 3. Multiple *C. virgata* on tree trunk. Photo courtesy of L. Poggiani, www.lavalledelmetauro.it

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NAPIS. National Pest Information System. http://napis.ceris.purdue.edu/

Cochlicella spp.

Scientific Name
Cochlicella acuta Muller, *Cochlicella barbara* L.

Synonyms:
Cochlicella ventrosa, *C. ventricosa*, *Helix acuta*, *Helix barbara*, *Helix bulmoides*, *Helix ventricosa*, *Helix ventrosa*, *Prietocella acuta*, *Prietocella barbara*, *Longaeva turrita*, *Xerophila cochleolina*

Common Name(s)
Pointed snail, conical snail, banded conical snail, small pointed snail, Mediterranean land snail, potbellied helicellid

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Stylommatophora, **Family:** Hygromiidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description **(from Kerney and Cameron, 1979)**
***Cochlicella acuta*:** 10-20 (rarely 30) x 4-7 mm. Shell a very elongated cone, with 8-10 slightly convex whorls with moderate sutures. Umbilicus minute, obscured by reflected columellar lip. Mouth elliptical, taller than broad, lacking internal rib. Shell white or ginger, often with darker bands and blotches, color and pattern very variable. Growth ridges irregular and rather weak. Prefers maritime habitat, usually in dunes and coastal grassland, occasionally calcareous ground inland.

***Cochlicella barbara*:** 8-12 x 5-8 mm. Shell is elongated cone of 7-8 very slightly convex whorls with shallow sutures. Umbilicus minute and partly obscured by columellar lip. Mouth elliptical and lacking internal rib. Shell thick and white, with some variation in color and banding as in *C. acuta*. Growth-ridges slightly more pronounced than in *C. acuta*, especially on last whorl. Prefers dry exposed sites near the sea, especially dunes.

Cochlicella are native to the western Mediterranean and were first reported in southern Australia in the 1920's (Baker, 1986). They are a particular problem in alfalfa, clover, wheat, and barley fields and pastures in Australia. *Cochlicella acuta* and *C. barbara* have been introduced into California and are currently established, where they can become locally abundant, often in greenhouse situations (Hardy, 2004).

Baker et al. (1991) studied the life history and population dynamics of *C. acuta* in a pasture-cereal rotation at Hardwicke Bay on Yorke Peninsula, South Australia. The life cycle of *C. acuta* was primarily biennial, with offspring being produced in large numbers in the pasture phase but not the cereal phase of the rotation. The breeding season lasts from autumn to spring. The one year old snails that infest crops in winter are slightly smaller in size (mostly 10-14 mm in height) than the two year snails that infest pastures at the same time (mostly 12-17 mm), but both groups have mature albumen glands suggesting they are both capable of reproduction. Snails were most abundant in the spring and summer, especially near the edges of fields. Snails aggregate on robust weeds such as yellow mignonette (*Reseda lutea*), as well as at the bases of grass tussocks and beneath loose rocks (Baker, 2002). An average of 7.1 clutches and 257.7 eggs per pair of *C. acuta* were produced during the laboratory breeding season (Baker and Hawke, 1991).

Figure 1. *Cochlicella acuta* (left) and C. *barbara* (right). Photos courtesy of SARDI. No scaling information available.

Cochlicella acuta and *C. barbara* are known to be an intermediate host of nematodes and trematodes which infect man and domestic animals (Godan, 1983; Morrondo et al., 2005).

Symptoms/Signs

Cochlicella acuta aestivates on the ears and stalks of cereals, contaminates grain (particularly barley) at harvest, clogs farm machinery, causes delays during harvest, causes damage to harvesting equipment. Contaminated grain can be cleaned mechanically but this is costly, because *Cochlicella* shells are

comparable in size to cereal grains (Baker, 2002). Snails cause feeding damage to crop seedlings and legume-based pastures and foul herbage with their slime. In Australia, contaminated grain and seed has been downgraded and rejected for export (Baker, 2002).

Survey

Preferred Method: The most effective method of survey for mollusks is through visual survey methods. While conducting a survey, look for snail eggs, juveniles and adults, as well as clues that suggest the presence of mollusk pests which may include: empty snail shells, mucus and “slime” trails, and/or ribbon-like feces.

On rainy or humid days, inspect containers that are suspended inches above the ground, garbage bins, driveways, low-growing bushes, and ground-lying trash. During dry or hot days, snails are attached, often in clusters of several to many individuals, to plants fences, and other objects; due to this behavior survey during dry weather may actually be easier, especially if snail density is low (CPHST, 2006).

In Australia, where the pest is known to be present, road sides are sampled extensively, because they provide favorable habitat and are easily accessible. Roads approximately parallel to each other and 8 km apart were plotted on large scale maps and sampling points (“stops”) were chosen at 8 km (5 mile) intervals along them (Pomeroy and Laws, 1967; Butler and Murphy, 1977).

Alternative Method: Soil litter can also be sampled to conduct a general mollusk survey. According to Nekola (2003), samples are collected and dried completely, then soaked in water for 3-24 hours, and finally subjected to careful but vigorous water disaggregation through a standard sieve series (9.5 mm, 2.0 mm. 0.85 mm, and 0.425 mesh screens). The fractions are then dried and passed through the same sieve series, and hand-picked against a neutral brown background. All shells and shell fragments are removed and identified.

At this time, a host risk map is available (Fig. 2). The host risk map describes the relative density (on a scale of 1-10) of susceptible hosts. The maps are based on National Agricultural Statistics Service (NASS) data. The scale of one to ten describes the proportion of total host acreage per county: for example a rank of one indicates no host acreage, while a score of ten indicates that 75-100% of the acres in the county contain suitable hosts for the pest. This map shows that portions of Arkansas, Colorado, Idaho, Illinois, Indiana, Kansas, Kentucky, Maryland, Minnesota, Missouri, Montana, Nebraska, North Dakota, Ohio, Oklahoma, Oregon, South Dakota, Tennessee, Texas, and Washington are at the greatest risk (rating of 4) from this mollusk based on host availability.

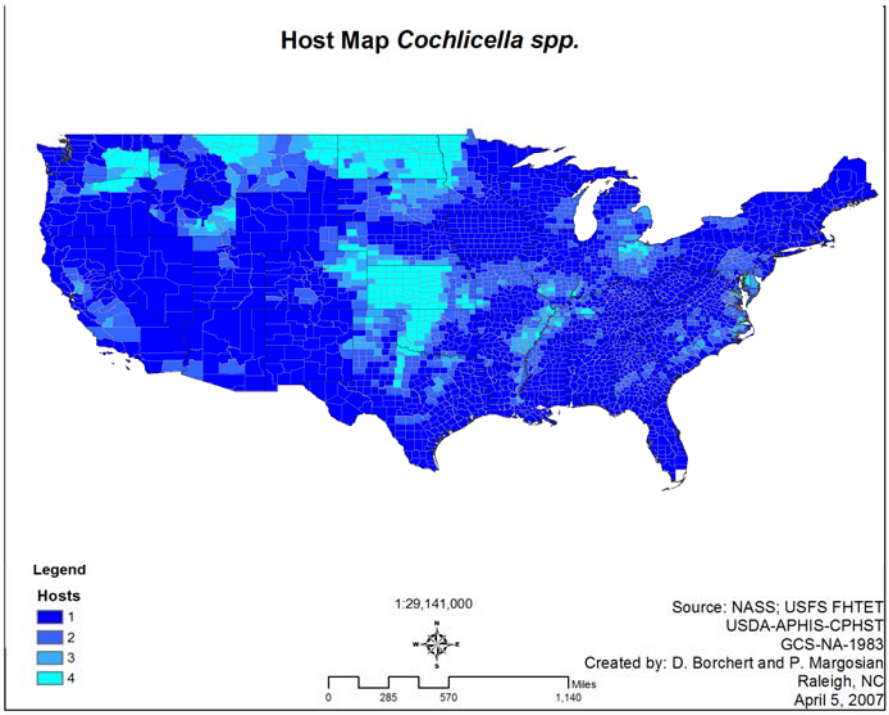

Figure 2. Host risk map for *Cochlicella spp.* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Note: Serious diseases are associated with the consumption and improper handling of certain mollusks (snails and slugs). Of particular concern, many mollusk species serve as intermediate hosts of nematodes and trematodes. While most cases of human infections result from consumption of raw or partially cooked snail meat, government inspectors, officers and field surveyors are at-risk due to the handling of live snail, samples, and potential exposure to mucus secretions. ***Wear neoprene gloves when handling mollusks and wash hands thoroughly after any mollusk survey or inspection activities.***

Key Diagnostics

Both species (*Cochlicella acuta* and *Cochlicella barbara*) of pointed snail are fawn or brown. The size and shape of the shell of mature specimens can be used to separate the two species. The shells of mature pointed snails are 12 to 18 mm long and the ratio of the shell length to its diameter at the base is always greater than two. The shells of mature small pointed snails are 8 to 10 mm long and the ratio of the shell length to its diameter at the base is always two or less (CABI, 2004).

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Tertiary Pests of Small Grains (Name and Photo only)
None at this time

Nematodes

Primary Pests of Small Grains (Full Pest Datasheet)

Heterodera filipjevi

Scientific Name
Heterodera filipjevi (Madzhidov) Stelter

Synonyms:
Bidera filipjevi sp. nov.

Common Name(s)
Cereal cyst nematode, Gotwald strain of *H. avenae*, pathotype 3 of *H. avenae*, race 3 of *H. avenae*.

Type of Pest
Nematode

Taxonomic Position
Class: Secernentea, **Order:** Tylenchida, **Family:** Heteroderidae

Reason for Inclusion in Manual
Requested by the CAPS community; found in Oregon in March 2008

Pest Description
Note: The cereal cyst nematodes consist of a complex group of closely related *Heterodera* species (*H. avenae*, *H. filipjevi, H. latipons*, and others), collectively known as the "*Heterodera avenae* group". There has been considerable disagreement with respect to the taxonomic classification of species in the *H. avenae* group. *Heterodera* species in the *H. avenae* group are distinguished from each other by small differences in detail rather than in gross morphology. Cereal roots infected by *H. filipjevi* show the same symptoms and lemon-shaped cysts as those

Figure 1. *Heterodera avenae* cysts attached to oat roots. Photo courtesy of R. Cook.

infected by *H. avenae* (Fig. 1). Only nematode taxonomists are able to identify this cyst nematode using a combination of morphological and molecular analysis.

Heterodera avenae, together with other bifenestrate cyst nematodes having a short vulval slit, were placed in the genus *Bidera*, but Mulvey and Golden (1983) synomized *Bidera* with *Heterodera* (Handoo, 2002). However, this synonomy was not universally accepted (Baldwin and Mundo-Ocampo, 1991).

Heterodera filipjevi is diagnosed by the following morphometrics (morphological measurements): Cyst length elongate 690 µm (490-830); bullae and underbridge present; vulval slit length 7 µm (6-8); second stage juvenile stylet length 27 µm (22-31) with slightly concave anteriorly directed knobs; tail length 57 µm (49-63); hyaline tail terminus length 35 µm (31-39) (Handoo, 2002).

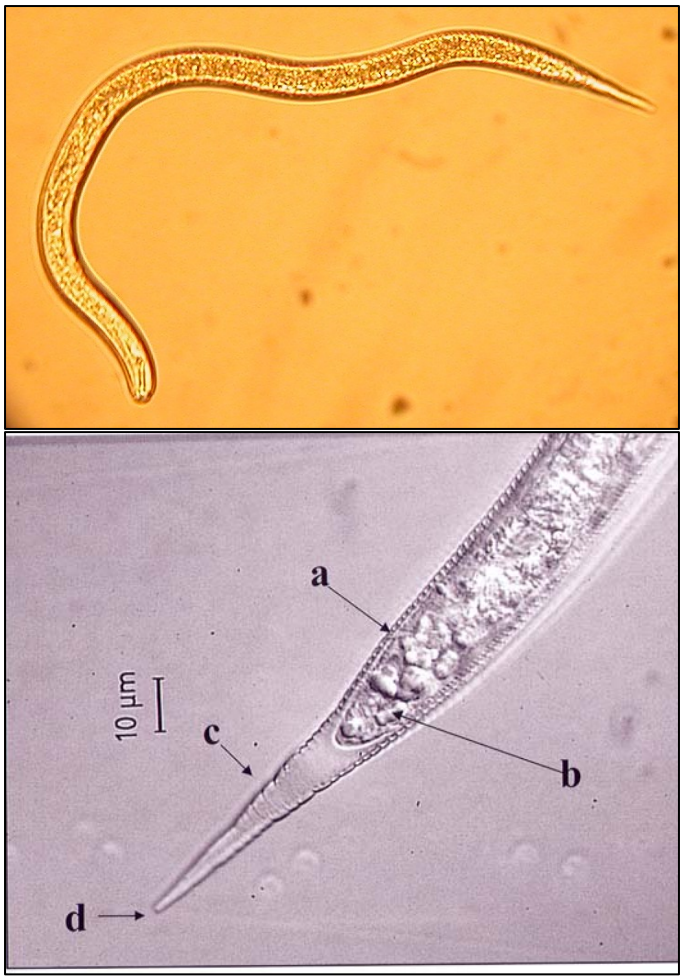

Figure 2. Top) *Heterodera filipjevi* J2. Bottom) Tail of *H. filipjevi* second stage juvenile with anus (a), true tail (b), hyaline tail (c), and the rounded tail tip (d). Photos courtesy of R. Smiley, Oregon State University and R. Holgado, Norwegian Institute for Agricultural & Environmental Research, Norway, respectively.

The second stage juveniles (Fig. 2) found in Oregon in 2008 (see Potential Distribution section) ranged from 530-570 µm in length, had a stylet (22.5-24.5 µm) with anchor-shaped basal knobs, and a tail (52.5-62.5 µm) with a hyaline tail terminal (30-38 µm) (Smiley et al., 2008). The lateral field had four lines of which the inner two were distinct.

The cysts were lemon-shaped, light brown in color, had a zigzag pattern on the cyst wall, and had a bifenestrate vulval cone with horseshoe-shaped semifenestra. The cysts were characterized by body length including neck 718-940 µm, body width 395-619 µm, l/w ratio of 1.1-2.2, neck length 75-140 µm and width 50-95 µm, fenestra length 50-65 µm and width 27-40 µm, heavy

underbridge 60-80 µm, vulval slit 7.5-8.5 µm and many bullae (Smiley et al., 2008).

Detailed descriptions for each life stage of *H. filipjevi* from Norway were provided by Holdago et al., 2004a, b.

Eggs: Cylindrical with rounded edges (Fig. 6) (Holdago et al., 2004b).

Second-stage juveniles (J2s): The second stage juvenile (Fig. 2) body length ranges from 455-557 µm, and the tail is tapering to a rounded tip. The length of the tail is 52-67 µm and its hyaline part measures 30-41 µm, corresponding to more than 50% of the total tail length. The head is offset and usually with three annules, and the distance from the head to the valves of the median bulb is 59-79 µm. The lateral fields have four lines, of which the inner two are more distinct, and the outer bands are heavily areolated (Fig 3). The stylet is robust with anchor-shaped basal knobs, and measures 22-25 µm in length (Fig. 3). The ratio of hyaline tail to the true tail is 1.2-1.7 (Holgado et al., 2004b).

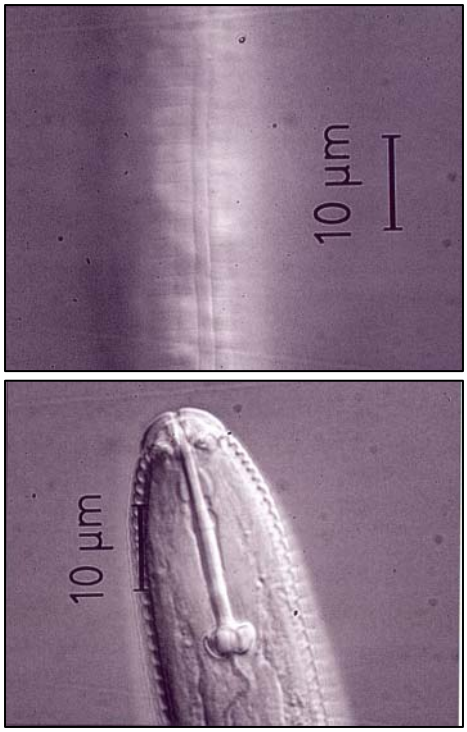

Figure 3. Top) The lateral field in the mid body region of *H. filipjevi* J2 has four lines with a heavy areolation on the outer bands. Note: the two distinct inner lines. Bottom) The stylet of *H. filipjevi* J2 is robust, with anteriorly concave knobs. Photo courtesy of R. Holgado, Norwegian Institute for Agricultural & Environmental Research, Norway.

Cysts: Newly formed cysts are lemon-shaped, similar to *H. avenae*, *H. latipons*, and *H. hordecalis* (Fig. 4, 5) and partially covered with a white sub-crystalline layer. The cyst wall has ridges running in zigzag patterns, and irregularly arranged punctations and pores. The cyst is golden to light warm brown and is almost transparent, with the outline of individual eggs clearly visible (Fig. 6). The cyst length is 455-874 µm and 253-747 µm in width. The vulval cone is bifenestrate with horseshoe-shaped semifenestra and has an underbridge. The vulval slit varies between 6.0 and 10.8 µm and the width of the vulval bridge is 7.2-13.1 µm. The fenestral length ranges from 38.4 to 58.4 µm and the length of the semifenestrae are 19.3-32.0 µm. The ratio between fenestral length and width was in the range of 1.7-2.8. The dimensions of the underbridge were 53-110 µm in length and 4.0-

11.3 µm in width. The bullae are weak to medium, distinct, and mostly globular in shape with a pale to medium brown color. Their position and arrangement vary between focal planes (Holgado et al., 2004b).

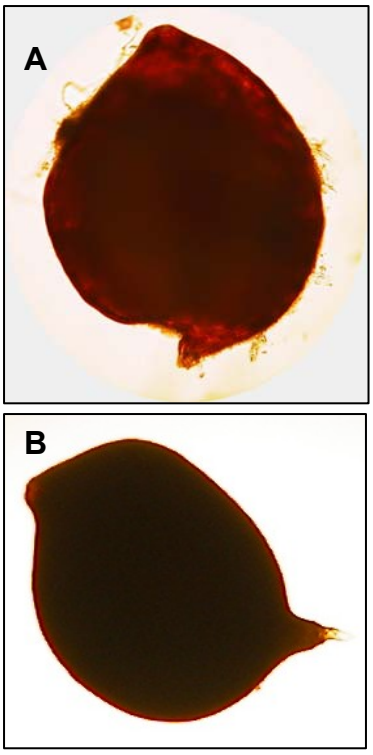

Figure 4. Cysts of A) *Heterodera filipjevi B) H. avenae*.Photos courtesy of R. Smiley, Oregon State University.

Females: Gravid females are pearly-white and lemon-shaped, with protruding neck and vulva cone. The cuticle bears a zigzag pattern that runs concentrically around the neck and vulval regions. The head is offset, with a squarish and prominent labial disc. The female stylet has sloping knobs. Ovaries are paired and convoluted. The vulva is slit-like, protruding posteriorly. The anus is distinct (Holgado et al., 2004b).

Biology and Ecology

The life cycle of *H.filipjevi* is essentially the same as for other species of *Heterodera*. Second-stage juveniles (J2s) hatch from the cyst (Fig. 7) at planting time and penetrate host roots just behind the root tip. The J2s emerge from the cysts, penetrate host roots, and establish a specialized feeding site (a syncytium) in the stele. They develop into swollen females, which retain the eggs and produce egg masses. Females rupture the root cortex and protrude from the root surface, leaving the head and neck embedded. Males, that have become worm-like or vermiform, move through the soil to inseminate females. When the female dies, the cuticle turns brown, and the body becomes a cyst filled with embryonated eggs, which develop to the second juvenile (J2) stage. The cysts break off from the roots and become free in the soil, where they perisist for many years and serve as the survival stage of the nematode. Like other cyst forming nematodes, it has sedentary endoparasitic habits. Because *H. avenae* has been reported to have only one generation per year (Nicol, 2002), it is assumed that *H. filipjevi* will have a similar number of generations per year. Information is not currently available on the persistence of cysts in the absence of host material. Information for *H. avenae*

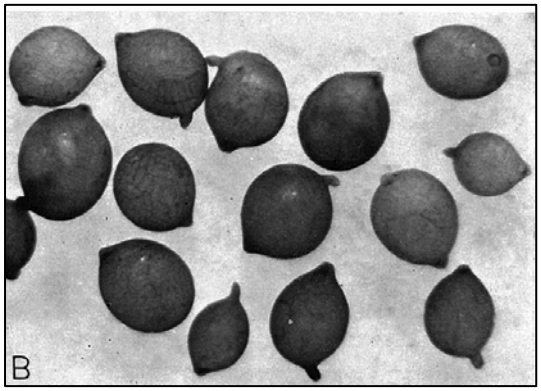

Figure 5. Lemon-shaped cysts of *H. hordecalis*. Photo courtesy of S. Andersson.

simply states that the eggs within the cysts remain viable in the soil for several years (Kort, 1972), and *H. filipjevi* will most likely be similar.

The greatest risk to cereal crops coincides with the presence of high populations of infective juveniles in the soil. This event occurs shortly after eggs hatch to release juveniles from inside the cyst. The juveniles move out of the cyst and into the soil where they may infest the tips of young root segments. *H. avenae* eggs hatch in early spring in Oregon (Smiley et al., 2007). Populations of *H. avenae* juveniles in the soil increase rapidly 2 weeks after mean weekly air temperature stabilized between 2-4°C (36 and 40°F). Peak populations of juveniles occur following a spike in weekly mean air temperature to 16°C (60°F) in mid-April.

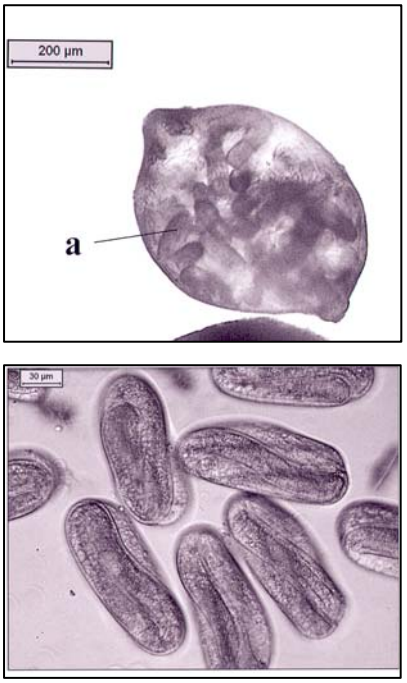

Figure 6. Top) The cyst wall of *H. filipjevi* is often transparent with the outline of eggs (a) clearly visible. Bottom) *H. filipjevi* eggs. Photos courtesy of R. Holgado, Norwegian Institute for Agricultural & Environmental Research, Norway.

According to Sahin et al. (2005), under controlled conditions optimum hatching for *H. filipjevi* occurred at 10°C (50°F 40.5%) and 18°C (64°F 42.8%) after 15 days. Hatching occurred but was significantly lower at 7°C (45°F 15.2%) and no hatch occurred at 0°C. Under field conditions, maximum hatch (~11%) occurred at soil temperatures of 3.7 and 8.8°C (39 and48 °F). Results in the field and laboratory did not correlate well, suggesting that factors other than temperature may be involved in the hatching of this species (*e.g.* host root exudates).

Because of this ability to hatch rapidly at cool temperatures, field population densities of *H. filipjevi* could easily be underestimated if based only on cyst extractions. Therefore, accurate assessments of pre-plant densities of *H. filipjevi* require the quantification of infective juveniles in the soil (Holgado et al., 2005). Sampling of juveniles, however, requires different soil sampling methodology than those required to sample cysts (Smiley et al., 2008).

Specialized virulence groups called pathotypes occur within species in the *H. avenae* complex and are roughly equivalent to the “race” concept used to define virulence groups for the pathogens that cause rust diseases. At least 12 pathotypes have been described for *H. avenae*. However, the Gotwald strain, race 3, and pathotype 3 of *H. avenae* have now been confirmed to have synonymy with *H. filipjevi*. Each pathotype is virulent to cereals unless a specific host resistance gene is present to nullify its virulence to that pathotype. A clear understanding of the pathotype identity is required before one or more genes for

resistance can be selected and used to improve the performance of wheat, barley, and oats planted on infested land (Smiley et al., 2007). Since *H. filipjevi* has been recently described as a species, there are no reports of pathotypes present within this species at this time.

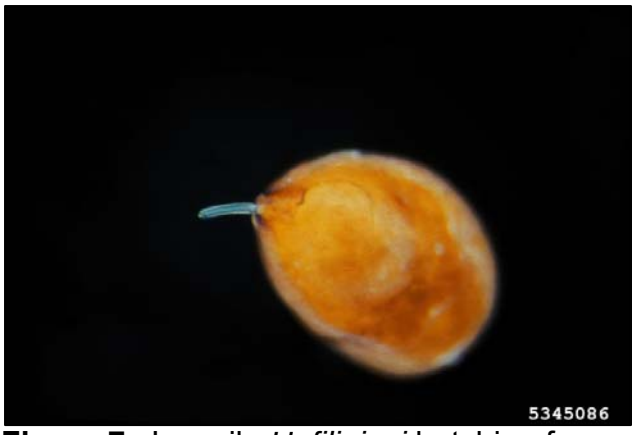

Figure 7. Juvenile *H. filipjevi* hatching from cyst. Photo courtesy of Bonsak Hammeraas, Bioforsk - Norwegian Institute for Agricultural and Environmental Research, Norway. www.bugwood.org

Symptoms/Signs

Symptoms are often due to mixed populations of cereal cyst nematodes or other soilborne pathogens and can mimic other problems such as nutrient deficiencies or drought stress.

In general, wheat plants become chlorotic and stunted due to the presence of cereal cyst nematodes. Growth may be patchy (Fig. 8). Barley roots exhibit no readily discernable symptoms. Leaf tips often become discolored: reddish yellow on wheat, red on oats, and yellow on barley (Smiley et al., 2007).

Figure 8. *H. filipjevi* damage on rye. Photo courtesy of Bonsak Hammeraas, Bioforsk- Norwegian Institute for Agricultural and Environmental Research, Norway. www.bugwood.org

In *H. avenae*, roots show abnormal branching or become bushy; this has not been observed with *H. filipjevi*. In Oregon, patches of stunted seedlings (3-5 leaf stage) appeared in March. The stunted seedlings exhibited chlorotic or necrotic lower leaves, healthy younger leaves, few or no tillers, rotting of lower culms and crown, and light brown roots with little or no branching (Smiley et al., 2008). In Oregon, *H. filipjevi* was found co-occurring with the fungal pathogens *Pythium* spp. (Pythium root

rot), *Gaeumannomyces graminis* var. *tritici* (take all of wheat), *Rhizoctonia solani* AG-8 (root rot, bare patch), and *Typhula incarnata* (snow mold) and the nematode species *Pratylenchus neglectus* (lesion nematode), and *Tylenchorhynchus* spp. (stunt nematodes). It is, therefore, difficult to determine which symptoms were caused by which pathogen.

Pest Importance

There is relatively little information on yield loss caused by H. *filipjevi,* perhaps because it was previously identified as *H. avenae.* However, with its increased prevalence, *H. filipjevi* is being recognized as a constraint to cereal production, particularly in temperate and semi-arid regions of the world (Nicol et al., 2004). In Norway, the occurrence of *H. filipjevi* was recorded recently causing damage to winter rye (Holgado et al., 2005), and this species seems to be an important pest of cereals in both Sweden and Norway (Ireholm, 1994; Holgado et al., 2004ab, 2005).

Known Hosts

Major hosts

Avena ludoviciana (wild oat), *Avena sativa* (oats), *Horedeum vulgare* (barley), *Secale cereale* (rye), *Triticum aestivum* (wheat) and other grasses, such as false wheat (*Elytrigia repens*) (Damadzadeh and Ansaripour, 2001; NPAG, 2008).

When plants were grown in plastic tubes, additional hosts were found (Bekal et al., 1998): *Triticum discocoides* (a tetraploid wheat), *T. durum* (a tetraploid wheat), *T. tauchi* (a diploid wheat), *T. monococcum* (a diploid wheat), *T. ovatum* (a tetraploid wheat), *T. tauchii* (a diploid wheat), *T. turgidum* (a tetraploid wheat), *T. umbellatum* (a diploid wheat), and *T. ventricosum* (a tetraploid wheat).

Zea mays (corn) has been reported a primary host and as a poor host. It appears that *H. filipjevi* is able to penetrate roots but females fail to reproduce. Corn has been used a trap crop in India (Bajaj and Kanwar, 2005).

Known Vectors (or associated organisms)

Heterodera filipjevi is not known to be a vector and does not have any associated organisms. However, root damage by cereal cyst nematodes often also favors greater colonization of roots by root-rotting fungal pathogens and by saprophytic bacteria, fungi, and non-plant-parasitic nematodes. These secondary organisms cause more intense rotting and discoloration than that caused by the plant-parasitic nematode itself.

Known Distribution

Because *H. filipjevi* was recently described as a distinct species and was also previously known as the “Gotland strain”, “race 3”, or pathotype 3” of *H. avenae*, there is some confusion concerning its actual distribution.

At present, the known distribution of *H. filipjevi* is as follows: **Asia:** India, Iran, Israel, Syria, Tadzhikistan, Turkey, and Uzbekistan. **Europe:** Bulgaria, England, Estonia, Germany, Italy, Norway, Poland, Russia, Spain, Sweden, and the Ukraine (Holgado et al., 2004a; Rumpenhorst et al., 1996; Nicol et al., 2004; Tanha-Maafi et al., 2007, NPAG, 2008).

The pathogen was confirmed to be present in the United States in March 2008.

Potential Distribution within the United States

On March 3, 2008, a nematode sample collected in northeastern Oregon from wheat was confirmed by the USDA-ARS Nematology Laboratory in Beltsville, MD as *Heterodera filipjevi*. This is the first report of the *H. filipjevi* in the United States (NPAG, 2008).

Biological information for this pest is not currently available to determine the potential distribution of this nematode within the United States. However, once a cyst nematode is introduced into a country, it is very difficult to minimize spread without extensive quarantine measures and expensive eradication strategies. Cyst nematodes are efficiently disseminated by all means of soil movement, including minute amounts of soil that contaminate equipment, by animals and plant products, and by soil that is moved by water and wind (NPAG, 2008). *Heterodera avenae* was first detected in the United States on oat in 1974 in Oregon. By 2007, *H. avena*e was present in seven states in the West: California, Colorado, Idaho, Montana, Oregon, Utah, and Washington (Smiley et al., 2007). Since *H. filipjevi* can hatch at temperatures at or below those of *H. avenae, a*reas of the United States that currently have *H. avenae* are at risk. Additionally, those areas that grow barley, wheat, rye, or oat are at risk that have spring temperatures from 3.7-18°C (48-64°F).

Survey

Preferred Method: Cereal cyst nematodes are extracted from soil, identified, and counted. A composite soil sample is collected from a sampling unit such as an entire field, a specific area of a field, or an experimental plot. Multiple (15 to 20) cores of soil are collected from the upper 8 to 12 inches of soil and combined for each composite sample. A subsample is removed from the larger sample and air-dried.

Cysts can be extracted from the subsample using a Fenwick can method (20 mesh screen over a 60 mesh screen) (Damadzadeh and Ansaripour, 2001), a modified Baermann funnel technique (Rumpenhorst et al., 2006), or a modified Fenwick can elutriation method (Fig. 9a) with further separation of cysts from the plant debris by flotation in an ethanol and glycerin solution (Fig. 9b) (Caswell et al., 1985; Ingham, 1994; Smiley et al., 2007). Cysts are then picked from the remaining debris and identified. Cysts can be crushed to determine the number of eggs plus juveniles, which can then be adjusted to reflect the nematode density per pound of oven-dry soil.

Survey for *H. filipjevi* should crossover easily and inexpensively if a state already has equipment to survey for potato cyst nematodes, because the Fenwick can method used to survey for *Globodera pallida and G. rostochiensis* should also work for the extraction of *H. filipjevi* cysts.

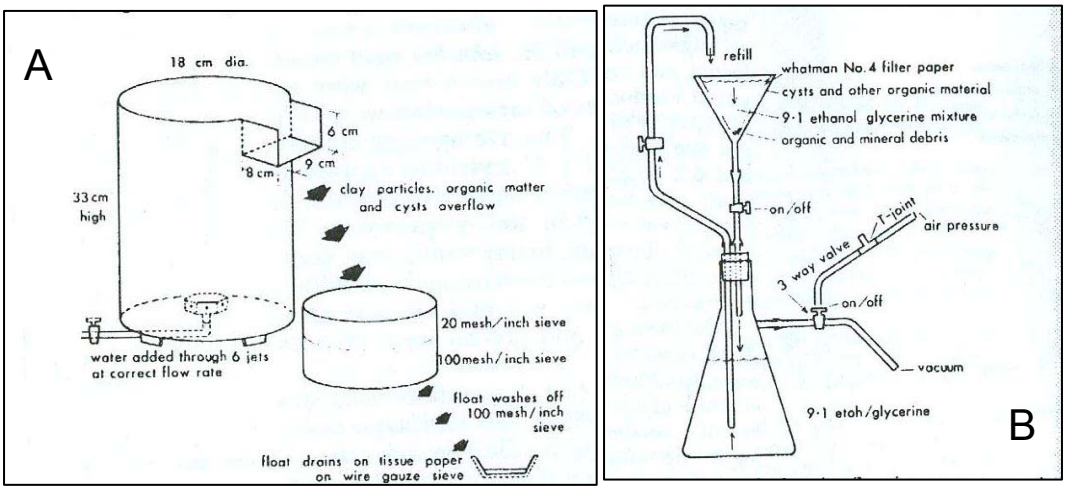

Figure 9. A) Modified Fenwick can used to separate nematode cysts and organic debris from soil sample B) Ethanol-glycerine flotation apparatus for separation of nematode cysts from sample organic matter. Reproduced from Caswell et al., 1985.

There a variety of additional methods available for cyst extraction from soil. Soil can also be processed using Cobb's decanting and sieving technique (10 mesh-2mm, 50 mesh-300 µm, 150 mesh-106 µm, 400 mesh-38 µm, and 500 mesh-25 µm screens used by Kanwar et al., 2004) and a 60 mesh sieve placed over a 300 mesh sieve used by Bajaj and Kanwar, 2005) to determine the presence of males, white females, and cysts. Subbotin et al. (2003) used sieving-decanting and centrifugation-flotation methods to isolate cysts from soil. Holgado et al. (2004b) air dried soil samples, passed the samples through a 5 mm sieve, and extracted cysts using a fluidizing column. Abidou et al. (2005) processed soil samples through a Kort elutriator.

Motile nematodes (*e.g.* juveniles) can be extracted using the Whitehead tray method (Smiley et al., 2008).

Key Diagnostics

Keys are available for identification by morphological characteristics of the cyst, second stage juvenile, male, and female. The vulval cone of *H. filipjevi* is bifenestrate and the semifenestrae are horseshoe-shaped (Fig. 10). The *H. filipjevi* vulval cone has a distinct underbridge (Fig. 10).

Handoo (2002) provides a key to the species within the *H. avenae* group as well as a thorough review of morphological studies to date. However, because of the

increasing number of species in the *H. avenae* complex, identification based on morphology is becoming more difficult (Subbotin et al., 2003). For precise identification, other methods are used such as (1) protein electrophoresis, (2) isozymes/ isoelectrofocusing (Andres et al., 2001), and (3) molecular biology techniques, such as restriction fragment length polymorphism (RFLP), random amplified polymorphic DNA (RAPD), and internal transcribed spacer ribosomal DNA (ITS-rDNA) (Bekal et al., 1997; Ferris et al., 1999; Andres et al., 2001; Nicol, 2002; Maafi et al., 2003; Rivoal et al., 2003; Subbotin et al., 1999; Subbotin et al., 2001; Subbotin et al., 2003; Madani et al., 2004; Abidou et al., 2005).

H. avenae, *H. filipjevi*, and *H. latipons* were also differentiated by electrophoresis on cellulose acetate plates using the enzymes esterase and malate dehydrogenase and aliquots of 25 females (Mokably et al., 2001).

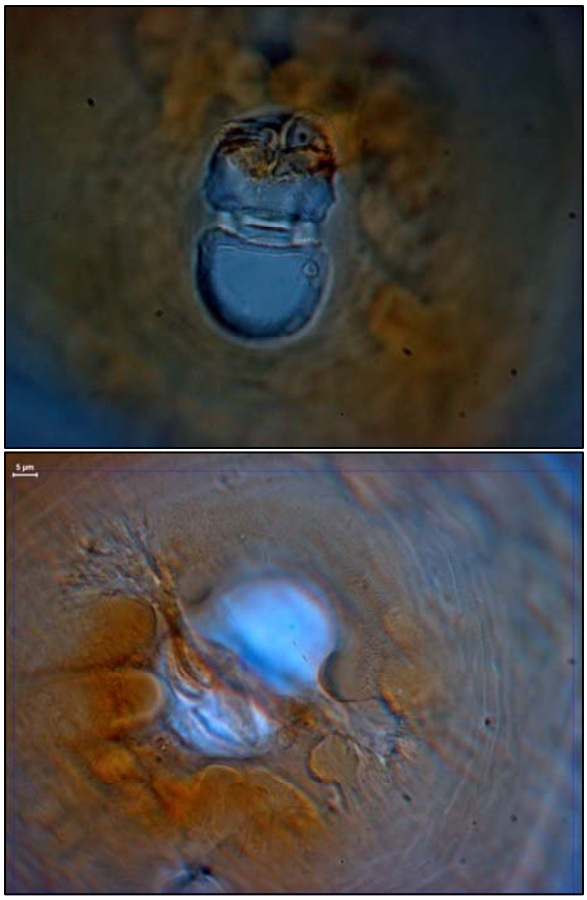

Figure 10. Top) Bifenestrate vulval cone of *H. filipjevi* with horseshoe-shaped semifenestrae Bottom) underbridge. Photos courtesy of R. Holgado, Norwegian Institute for Agricultural & Environmental Research, Norway.

Easily Confused Pests

H. filipjevi has been confused with several other cyst nematode species that parasitize cereals, including (but not limited to) *H. avenae, H. bifenestra, H. hordecalis, H. latipons*, *H. mani, H. pakistanensis, H. tucomanica,* and *H. zeae* (Kort, 1972; Nicol, 2002).

H. filipjevi and *H. avenae* can be separated on the basis of morphology of the cyst (Fig. 4, 6) and the vulval cone (Fig. 10,11) (Holgado et al., 2004b). *H. filipjevi* has a well developed underbridge, whereas the underbridge in *H. avenae* is weakly developed or absent. Bullae (blister-like prominences near the vulval fenestra) in *H. filipjevi* are weak to medium, distinct, mostly globular and pale to medium brown in color, whereas they are a strong, dark brown, numerous, clearly distinct, and variable in shape in *H. avenae*. The cyst wall of *H. filipjevi* is light in color, but those of *H. avenae* have dark brown to black cyst walls. Eggs of

H. filipjevi are easily observed through the cyst wall, unlike those of *H. avenae* (Fig. 6) (Holgado et al., 2004b).

H. latipons cysts differ from those of *H. filipjevi*, because they lack bullae which are present on those of *H. filipjevi* (Greco et al., 2002).

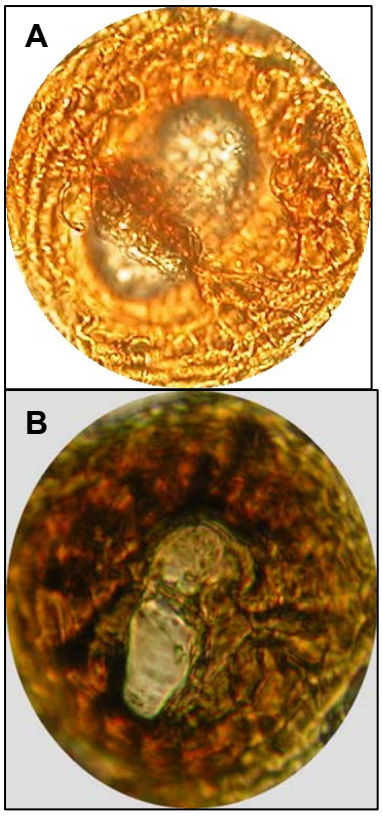

Figure 11. Vulval cones of A) *Heterodera filipjevi* and *B) H. avenae*. Photos courtesy of R. Smiley, Oregon State University.

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Heterodera latipons

Scientific Name
Heterodera latipons Franklin

Synonyms:
Bidera latipons, Ephippiodera latipons

Common Name(s)
Mediterranean cereal cyst nematode

Type of Pest
Nematode

Taxonomic Position
Class: Secernentea, **Order:** Tylenchida, **Family:** Heteroderidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description
Note: There has been considerable disagreement with respect to the taxonomic classification of *Heterodera latipons* and closely associated species referred to as the "*H. avenae* group". *H. avenae*, together with other bifenestrate cyst nematodes having a short vulval slit, were placed in the genus *Bidera* (Handoo, 2002), but Mulvey and Golden (1983) synomized *Bidera* with *Heterodera*. This synonomy was not universally accepted (Baldwin and Mundo-Ocampo, 1991; Davis and Venette, 2004).

(From Franklin, 1969; Greco et al., 2002)
Eggs: length 100-124 μm; width 44-56 μm.

Second-stage juveniles (J2s): Body length 401-478 μm; body width 19-22 μm; tail length 42-54 μm; length of the hyaline tail tip 20-31 μm; stylet length 23-25 μm; laterial field with four incisures.

Cysts: Cysts are typically ovoid to lemon-shaped as those of *H. avenae* with short vulva slits (< 16 μm). Fenestral length 58-76 μm; fenestral width 15-27 μm; semi-fenestral length 13- 19 μm; vulval slit length 6-9 μm; vulval bridge length 18-39 μm; underbridge length 80-125 μm; underbridge width 7-14 μm; sub-crystalline layer present

Females: Body length (excluding neck) 348-645 μm; body width 277-510 μm; neck length 58-103 μm; stylet length 21-28 μm

Males: Body length 960-1406 μm; body width 25-32.5 μm; stylet length 22-29 μm; spicule length 32-36 μm; lateral field with four longitudinal incisures.

Biology and Ecology
The life cycle of *H. latipons* is essentially the same as for other species of *Heterodera*. Second-stage juveniles (J2s) hatch from the cyst at planting time and penetrate host roots just behind the root tip. The J2s emerge from the cysts, penetrate host roots, and establish a specialized feeding site (a syncytium) in the stele. They develop into swollen females, which retain the eggs and produce egg masses. Females rupture the root cortex and protrude from the root surface leaving the head and neck embedded. Males that have become worm-like (vermiform) move through the soil to inseminate females. When the female dies, the cuticle turns brown, and the body becomes a cyst filled with embryonated eggs, which develop to the second juvenile stage. Cysts persist in soil for many years and serve as the survival stage of the nematode.The cysts break off from the roots and become free in the soil (USDA, 1985). Like other cyst forming nematodes, it has sedentary endoparasitic habits.

Laboratory experiments demonstrated that eggs hatch in greater numbers from older (4-5 months) than from younger (1-2 months) cysts after exposing the cysts to temperatures of 5°C or 10°C. Temperatures in the range 5-15°C appear to be the most suitable for egg hatch, while at 20-25°C egg hatch seems to be suppressed. Field studies under Mediterranean conditions indicate that nematode J2s occur in the soil from November to February when soil temperatures do not exceed 18°C. Usually, J2s in the soil peak at plant emergence Juveniles invade roots behind the root apex (at root apex in *H. avenae*) at the beginning of plant emergence in November-December.

Females and males develop by the end of January, and females lay eggs by February. Embryonated eggs can be observed by early March, when well-developed white females can be easily observed on the roots. From April onwards, white females turn to brown cysts coated by a sub-crystalline layer. In cooler areas, as in inland Syria, the development of the nematode can be delayed. Accumulated day degrees, above 7°C, for the development of white females and cysts with coiled embryos were 215 and 386, respectively. Only one generation per growing season is completed (Greco et al., 2002).

Symptoms/Signs
Slight to severe yellowing of cereal stands can be observed at an early stage of nematode infestation. Later, infested fields show patchy plant growth associated with poor tillering and shorter spikes. Symptoms occur in patches that enlarge as the nematode population increases. These symptoms are similar to those caused by other biotic and abiotic stresses. Before plant flowering, white lemon-shaped females can be observed on the roots by the naked eye or under a dissecting

microscope after gently shaking or washing the roots to remove adhering soil (Greco et al., 2002). Plants also tend to wilt during warmer portions of the day

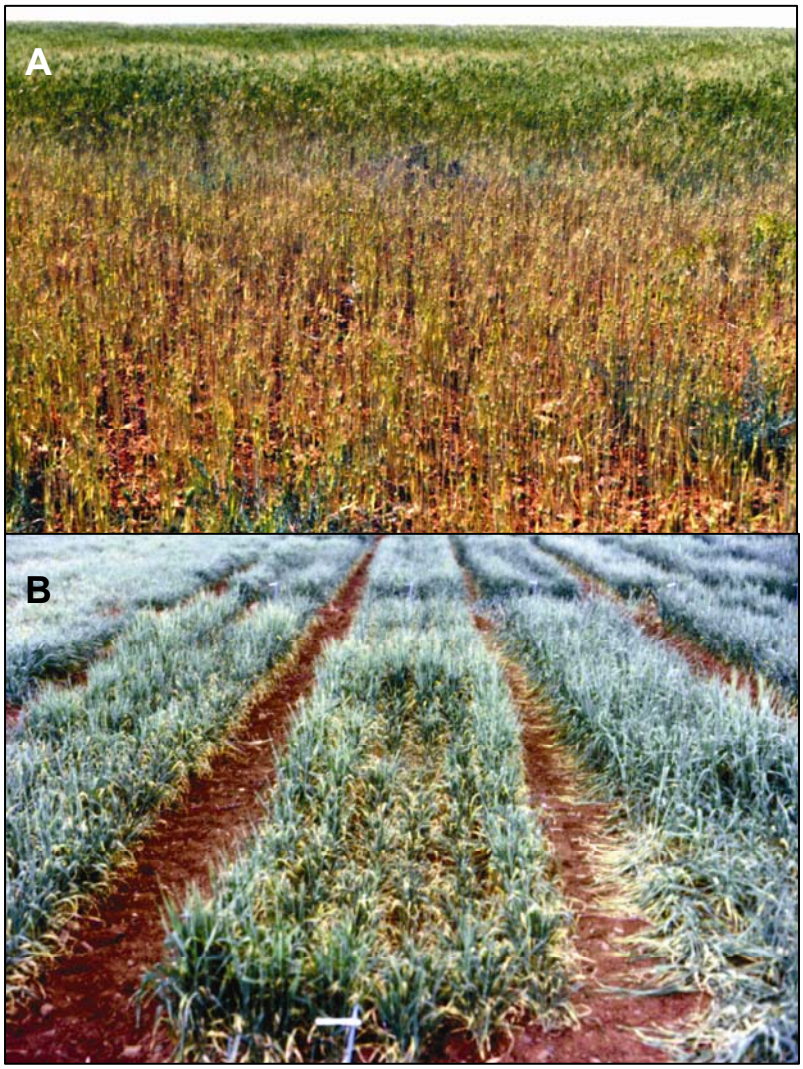

Figure 1. Symptoms of infection by *Heterodera latipons* in: A) Syrian durum wheat and B) Cyprus barley. Photos courtesy of R.N. Inserra from Greco et al. (2002).

Pest Importance

Although severe damage has been observed, no investigation has been undertaken to relate nematode population densities with yield of host crops. Therefore, little information is available on the extent of yield losses caused by *H. latipons*. In addition, the economic importance of *H. latipons* is difficult to assess because it has been confused with *H. avenae* and in some places, occurs in mixed populations with *H. avenae* (USDA, 1985). Philis (1988) reported up to 50% yield loss of barley in Cyprus. A 24% yield loss was observed in barely infested with 28 eggs and juveniles/g of soil located in an area with 279 mm

annual rainfall, but no significant yield reduction was observed in soil infested with up to l0 eggs/ g soil in areas with 411 mm annual rainfall (Greco et al., 2002). These findings would confirm field observations that the nematode is more damaging under water stress conditions.

Known Hosts

Ammophila arenaria (marram grass), *Arachis hypogaea* (peanut), *Avena sativa* (oats), *Beta vulgaris* ssp. *vulgaris* (beet), *Daucus carota* (carrot), *Hordeum vulgare* (barley), *Phalaris* spp. (canarygrass), *Secale cereale* (rye), *Triticum aestivum* (wheat), *Triticum durum* (wheat, durum), and *Triticum spp.* (wheat). *Triticum durum* is reported as a poor host (Greco et al., 2002).

Barley and wheat are the most seriously damaged cereals, but the nematode also infects oats and rye. Carrot may also be a poor host as *H. latipons* was reported at low population densities and without causing notable damage (Tacconi, 1976). No other authors have reported carrot as a host (Davis and Venette, 2004). Mackintosh (1970) reported marram grass as a host of *H. latipons*; however, this was probably a misidentification

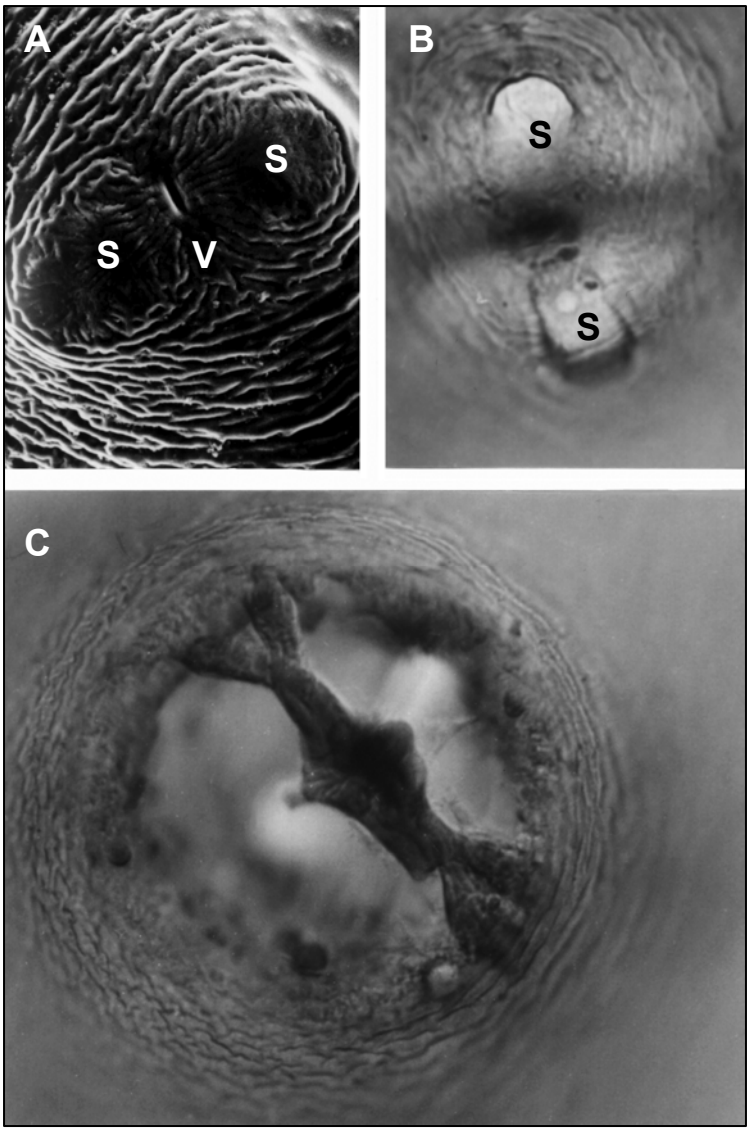

Figure 2. Micrographs and photographs showing the anatomical characteristics of *H. latipons* cysts. A) Scanning electron micrograph of the vulval area of a newly formed cyst. Note: the short vulval slit (v) and the area in which fenestration (S) will occur at a late stage of cyst formation. B) Light microscopy photograph of the perineal pattern of a nematode cyst showing well-separated and circular semi-fenestra. C) Perineal patterns observed with a light microscope showing a well-developed and a rather long underbridge, bifurcated at both ends and with a sclerotized enlargement in the middle. Photos courtesy of R.N. Inserra from Greco et al. (2002).

of *H. hordecalis* (Davis and Venette, 2004).

Known Vectors (or associated organisms)

Damage is more severe in fields infested concomitantly by *H. latipons* and the fungus *Bipolaris sorokiniana* (Sacc,) Shoemaker, the causal agent of common root rot and seedling blight of barley, as the nematode increases the aggressiveness of the fungus (Greco et al., 2002).

Known Distribution

Asia: Armenia, Iran, Israel, Japan, Jordan, Syria, Tajikistan, Turkey, and Turkmenistan. **Africa:** Algeria, Libya, South Africa, and Tunisia. **Europe:** Bulgaria, Cyprus, former Czechoslovakia, Greece, Italy, Poland, Spain, Slovenia, Scotland, and Ukraine. **North America:** Canada (CABI, 2004; Davis and Venette, 2004; Greco et al., 2002).

Reports from Prince Edward Island, Canada and Scotland, however, are likely *H. hordecalis* (USDA, 1985).

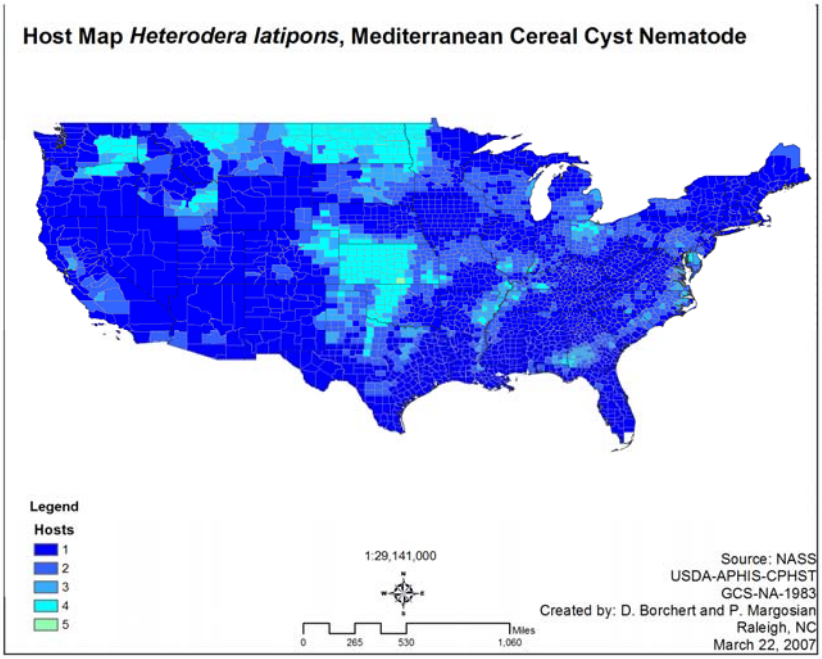

Figure 3. Host risk map for *H. latipons* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Potential Distribution Within the United States

Considering the large variety of climatic conditions under which *H. latipons* has been reported and its temperature requirements for development, there is no

doubt that this cyst-forming nematode would become a very noxious pest for winter cereals, especially in temperate states in the United States (Greco et al., 2002). At this time, a host risk map is available (Fig. 3). The host risk map describes the relative density (on a scale of 1-10) of susceptible hosts. The maps are based on National Agricultural Statistics Service (NASS) data. The scale of one to ten describes the proportion of total host acreage per county: for example a rank of one indicates no host acreage, while a score of ten indicates that 75-100% of the acres in the county contain suitable hosts for the pest. This map shows that Sumner Co. Kansas has the greatest risk of *H. latipons*. Portions of Alabama, Arkansas, Colorado, Georgia, Idaho, Illinois, Indiana, Kansas, Kentucky, Missouri, Montana, Nebraska, North Dakota, Oklahoma, Ohio, Oregon, South Dakota, Texas, and Washington where small grains are grown are at risk (ranking of 4) from this nematode based on host availability.

Survey

Preferred Method: As for other cyst-forming nematodes, in the absence of a host crop, soil samples must be collected and processed to extract cysts. Cyst nematodes are often extracted from soil using some form of elutriation or flotation. The Fenwick flotation can is frequently used for this purpose (Davis and Venette, 2004). Perineal patterns of cysts must then be prepared, observed under a microscope and compared with those of the original description (Franklin, 1969) or processed using molecular analyses. In the presence of a host crop (winter cereals), close examination of roots will reveal the presence of white lemon-shaped females. However, at a later stage of development, females become easily detached from the roots making the detection of the nematode very difficult even in cases of heavy infestation, especially in non-sandy soils. Roots of cereals infested by *H. latipons* do not show the typical branching of those infested by *H. avenae*. Moreover, females of *H. latipons* are rather isolated while in *H.avenae* they tend to be grouped (Greco et al., 2002).

In the field, the most reliable method for detection is the collection of soil samples in a grid pattern and processing by a wet screening method (USDA, 1985). For identification, a minimum of 10 cysts with juveniles is desirable. Males and females will help in identifying field infestations (USDA, 1985). Davis and Venette (2004) give considerable detail about soil sampling (e.g., number of samples to take, subsampling, and factors that influence efficiency of this method).

Key Diagnostics

Heterodera latipons cysts are typically ovoid to lemon-shaped as those of *H. avenae.* They belong to the *H. avenae* group because they have short vulva slits The fenestration of *H. latipons* cysts shows two distinct semi-fenestrae, which are more than a semifenestral width apart. The underbridge is strong and shows a pronounced thickening in the middle from which the name of the species is derived. The extremities of the underbridge are bi-trifurcate. Bullae are few to absent (Fig. 2) (Greco et al., 2002).

Cyst, J2, male and female morphology provides diagnostic information for the identification of this nematode species (Handoo, 2002). Handoo (2002) provides a key to the species within the *H. avenae* group as well as a thorough review of morphological studies to date. Greco et al. (2002) give detailed information on the morphological differentiation of four *Heterodera* species and include scanning electron micrographs of key anatomical characteristics.

Easily Confused Pests

Heterodera latipons may occur by itself or in mixed populations that include closely related *H. avenae* or *H. trifolii*. *H. latipons* has been confused with several other cyst nematode species that parasitize cereals, including (but not limited to) *H. avenae, H. bifenestra, H. filipjevi, H. hordecalis, H. mani, H. pakistanensis, H. torcomanica, H. zeae*, and a more taxonomically distant species, *Punctodera punctata* (Kort, 1972; Nicol, 2002). Franklin (1969), who first described *H. latipons*, compared morphological characters of *H. avenae, H. tucomanica* and *H. latipons.* Due to technological advances in molecular diagnostics, differentiating among morphologically similar cyst nematodes can be completed most reliably by restriction fragment length polymorphism (PCR-RFLP) of ribosomal DNA (Bekal et al., 1997; Subbotin et al., 1999; Subbotin et al., 2001; Handoo, 2002; Nicol, 2002; Maafi et al., 2003; Rivoal et al., 2003; Madani et al., 2004).

Heterodera avenae, *H. filipjevi*, *H. latipons* and *H. mani*, were also differentiated by electrophoresis on cellulose acetate plates using the enzymes esterase and malate dehydrogenase and aliquots of 25 females (Mokably et al., 2001).

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Meloidogyne artiellia

Scientific Name
Meloidogyne artiellia Franklin

Synonyms:
None

Common Name(s)
British root-knot nematode, cereal and legume root-knot nematode

Type of Pest
Nematode

Taxonomic Position
Class: Secernentea, **Order:** Tylenchida, **Family:** Heteroderidae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description (see Figure 1)

From Franklin (1961, 1978):
Eggs: (n=20), length 75-111 µm; breadth 34-43 µm.

Juveniles: (n=10-20), body length 301-370 µm; body breadth 10-16 µm; tail length 18-26 µm; stylet length 14-16 µm. The most striking feature of the larvae is the short tail with rounded tip. It is about 24.5 µm long, and two and one half times as long as the body diameter at the anus.

Females: (n=8-10 specimens), length 650-760 µm; width 340-460 µm; stylet 12-16 µm; vulva 15-22 µm.

Body swollen, pear- or flask-shaped, tapering gradually anteriorly to a small head; smooth, rounded posteriorly, with terminal vulva. Annules visible in neck region and around tail. The broad “neck” narrows abruptly at the head which is 4-5 µm across. In face view, there appear to be six almost equal lips, and a small labial cap around the mouth aperture. The amphids open as short slits on the inner edge of the lateral lips. Each of the four sub-lateral lips has a small papilla, but none was visible on the lateral lips. Optical sections show a delicate, six-radiate skeletal structure around the anterior end of the stylet, but it disappears below the level of the lips. Dorsal views of the head show a constriction on the lateral lips about one-third behind the anterior edge. These lips could, therefore, be described as consisting of two unequal annules. The excretory pore lies ventrally one or two stylet lengths behind the head. The cuticular (perineal)

pattern around the vulva and anus is characteristic. It is formed of striae and ridges of the cuticle, the latter being more pronounced nearer the vulva an anus. In general outline, the pattern is roughly that of a figure eight, the upper, smaller area enclosing the phasmids which are usually quite distinct, the anus situated at the center and the vulva occupying the diameter of the lower, larger part of the pattern. At the top of the arch, which is morphologically the dorsal part of the tail, the pattern is usually angular. Cuticular folds curve towards the anus from each side but leave a smooth unpatterned area around the vulva. The vulva is further from the anus in relation to the tail length than in most other species of the genus. The distance from the anus to vulva is about three times that from the anus to a line joining the phasmids. The exact position of the tail tip is difficult to determine because the lateral lines are marked only by the position of the phasmids and by slight irregularities in the striae.

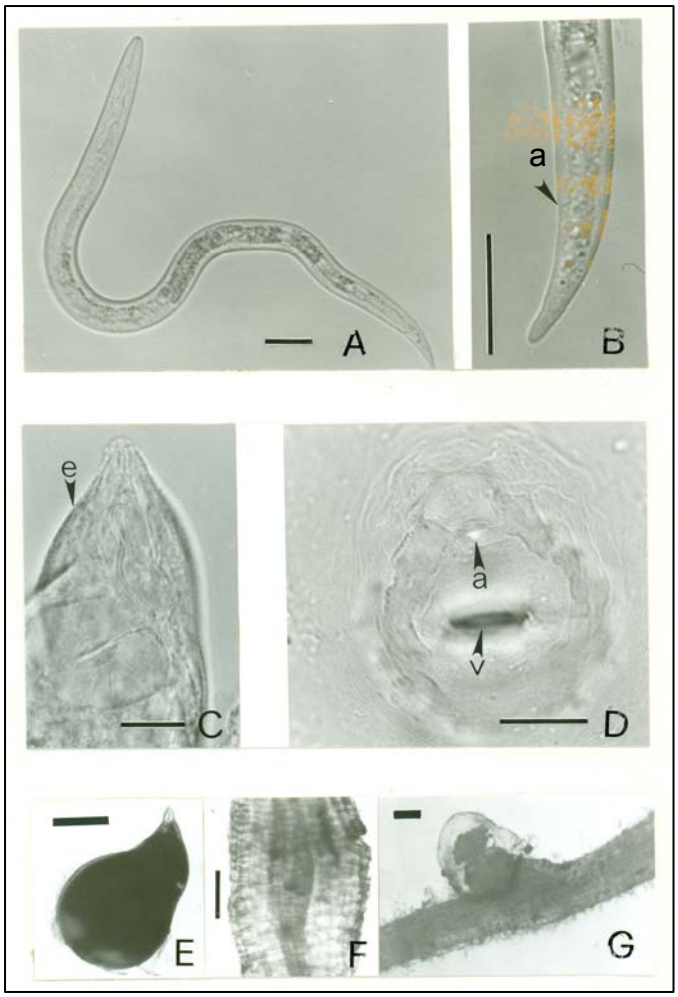

Figure 1. Photomicrograph of *M. artiellia* life stages. Scale bars = 20 µm in A-D and 200 µm in E-G. A) entire body of second-stage juvenile (J2); B) posterior body portion of J2, a=anus; C) anterior body portion of swollen female, e=excretory pore; D) perineal pattern showing the eight-shaped inner area marked by coarse lines and containing vulva (v) and anus (a), Note the fine striae and continuous striae surrounding the inner area; E) entire body of swollen female; F) slight swelling induced by J2 on chickpea root; G) large egg mass covering a swollen female, which protrudes with its posterior portion of the body from the surface of a chickpea root. All photos courtesy of R.N. Inserra.

Males: (n=7-15), length 0.82-1.37 µm; width 23-36 µm; stylet 17-27 µm; a=31-40; *b=10-15; c=60-100, where a=length/greater diameter; b=length/distance from head end to end of oesophagus; c=length/length of tail (anus to tip)] *Measurements for b were made from the anterior end to the posterior edge of the

oesophageal bulb, as the end of the glandular region overlaps the intestine and is difficult to define.

Body annulated, annules about 1.5 µm wide. Lateral fields with four incisures at the tail, but along the greater part of their length a fifth incisure is present in the center of each field. The lateral fields continue round the tail which is twisted through about 90º. Phasmids small, approximately adanal. Head with labial cap and six nearly equal lips. Face views show the slit-like amphid openings on the lateral lips; papillae not seen, nor was the stellate skeletal structure, such as that in the female. In dorso-ventral view a constriction is seen on the lateral lips about one-third from the front. A tubular guide surrounds the anterior end of the stylet which has well-developed, rounded, basal knobs. Pro-corpus narrow, two to three body-widths long, followed by a spindle-shaped muscular corpus about twice as long as wide. The oesophageal glands stretch for about three body-widths ventro-laterally along the intestine. Nerve ring one bulb-length behind muscular bulb. Two body-widths behind the oesophageal bulb is a conspicuous hemizonid and immediately behind it is the excretory pore with its duct running back for a short distance. Spicules typical for the genus, curved with anterior thicker part and tapering posteriorly to a point. A small gubernaculum, about one-third the length of the spicules, lies dorsally in the cloaca wall. Tail very slightly longer than the anal body diameter.

Biology and Ecology

Di Vito and Greco (1988a) have investigated the biology of *M. artiellia* on chickpea under Mediterranean climate conditions using growth chamber and microplot studies. Like several nematodes, *M. artiellia* is adapted to cool and dry conditions and has the ability to enter into an inactive, quiescent state to survive environmental stresses (Jensen, 1972). In climates with cool, wet winters and warm, dry summers, *M. artiellia* is active during spring and winter months and inactive from late spring through summer. Typically one generation is completed under non-irrigated conditions.

Development time depends on temperature. Temperatures of 10ºC and 30ºC have been reported as unfavorable for root penetration, development and egg production, while temperatures in the 15-25ºC range are considered optimal. *M. artiellia* has a reported threshold temperature of 10ºC.

This species reproduces asexually, but has the ability to produce sexually when conditions are appropriate (DeGiorgi et al., 2002). Adults have reportedly occurred 14-18 days following root penetration at optimal temperatures between 15-25ºC. Females develop after 230-240 degree days over 10ºC. Females swell, producing large gelatinous egg masses or sacs, containing between 500-1000 eggs. The egg sac is deposited on either galled root surfaces or inside root galls (Hussey, 1985).

Egg hatch may or may not involve stimulation from the host root (Hussey, 1985).

Hatching can occur for an extended period at temperatures between 5-10ºC. Hatch is most rapid at temperatures between 15-25ºC. Eggs will not hatch under extended dry periods but may persist in soil or dry roots awaiting more favorable moist soil conditions. Emergence occurs under moist soil conditions; juveniles may become inactive under dry conditions.

Meloidogyne larvae and eggs can be easily distributed by irrigation ditches, and in areas of saturated soil, larvae may survive under water for up to three weeks (Milne, 1972). *M. artiellia* can also reportedly survive in "fallow" fields for 1-2 years (Jensen, 1972).

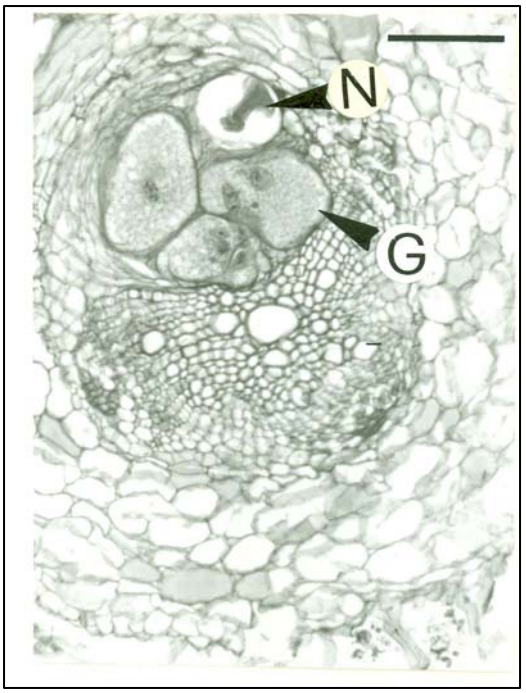

Figure 2. Cross section of a chickpea root infected by *M. artiellia*. Scale bar = 70 µm. Note: the specialized giant cells (G) which provide nutrients to the nematode (N).

There are four juvenile stages. The first stage occurs inside the egg. Following a molt and emergence, second stage juveniles move out of the egg and invade the host plant roots (Hussey, 1985). The second is the only stage when juveniles are mobile and are thought to be attracted to host plant roots (Hussey, 1985). They may feed singly or in a group.

If a larva cannot find a suitable feeding site on a host, it will continue searching until its energy is depleted. When a suitable site is selected, the larva will penetrate the root, usually near or behind the root cap, at lateral root initials or in galled root tissue near an embedded adult female. The site where one juvenile enters the root may attract others (Hussey, 1985). The juvenile moves through the root to the region of cell differentiation, settles, and becomes inactive while feeding.

Feeding induces cells in the primary phloem or parenchyma to swell and form "giant" or "nurse" cells on which juveniles feed until development is complete (Hussey, 1985) (Fig. 2). If the plant does not form giant cells as the nematode attempts to establish a feeding site, the larva may not complete its development and leave in search of another root, or die of starvation in the process (Jensen, 1972; Hussey 1985). When giant cell formation occurs, tissues surrounding the feeding nematode begin transforming at approximately the same time, producing a gall within 1-2 days following root penetration (Hussey, 1985).

A female larva will swell as it feeds until development is completed. Total development time varies from approximately 20 days at 25ºC to 55 days at 10ºC. Following chickpea root penetration, third and fourth stage juveniles have been observed 3-5 and 10-12 days, respectively, at 15-25ºC (Di Vito and Greco, 1988a).

Symptoms/Signs

Damage to host plants caused by root-knot nematodes involves impaired root growth (e.g., small gall formation (Fig. 3), proliferation of lateral roots, or stimulation of giant cell growth at feeding sites in parenchyma and phloem) and impaired root function (contributing to chlorosis, stunted growth (Fig. 3), nutrient deficiencies, and/or necrosis of above-ground plant parts). Symptoms of nematode damage may be similar to those caused by nutrient or water deficiency.

Nematode infestation of plant roots limits water uptake. Infested plants may appear wilted under hot and sunny conditions, even with ample soil moisture. Symptoms may not be apparent until plants reach later stages of growth.

Injured root tissue is susceptible to other disease-causing

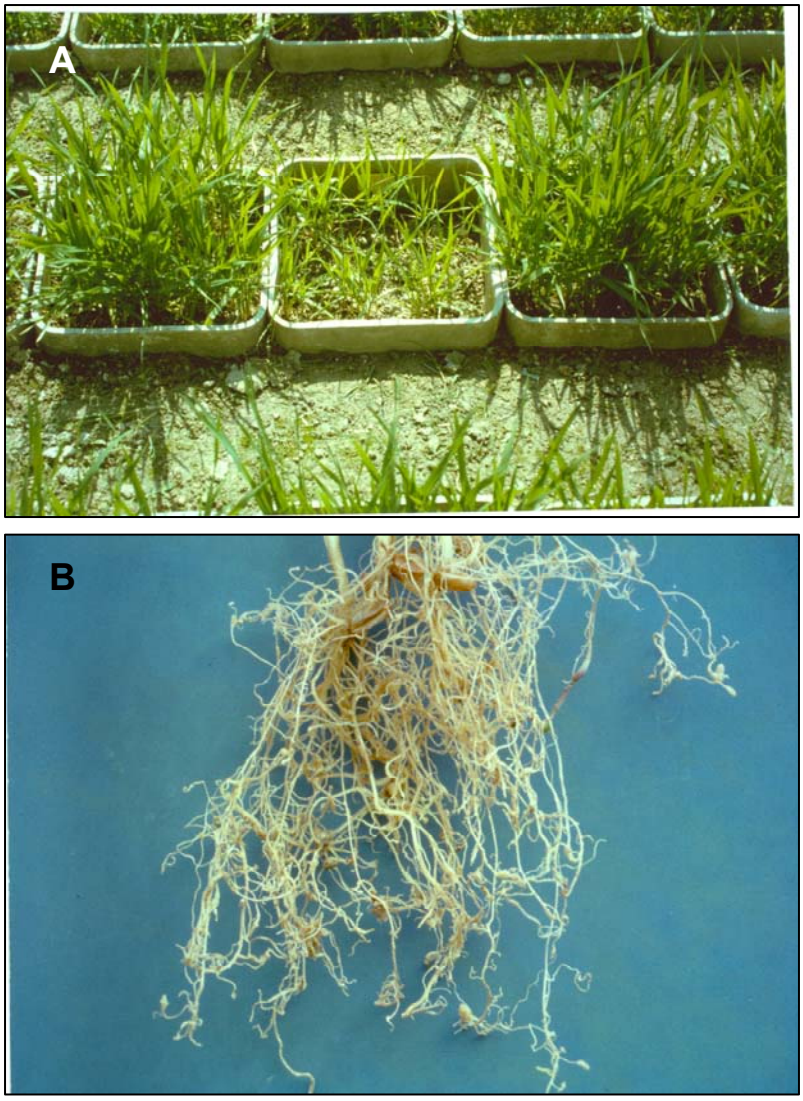

Figure 3. A) Stunted hard wheat plants in a microplot heavily infested by *M. artiellia* (center) compared to healthy plants in a non-infested control plot (right) and a plot slightly infested with the nematode (left). B) Hard wheat roots infected by *M. artiellia*. Note: the small galls and root proliferation induced by the nematode infection. All photos courtesy of R.N. Inserra.

pathogens. Much of the visible damage to plant hosts is likely caused by a combination of biotic and abiotic factors (Davis and Venette, 2004).

In wheat, spikes are sparse and reduced in size. Root galls induced by *M. artiellia* are very small and often are covered by large egg masses that represent the only visible signs of the nematode infection (Greco et al., 1992).

Pest Importance

M. artiellia has been reported as a damaging pest of cereals and leguminous crops in European and Middle Eastern countries. Consistent damage on chickpea has been observed in fields after rotation with wheat. Damage to wheat has been reported in Mediterranean countries where wheat is continuously grown (Di Vito and Greco, 1988a). Microplot studies indicate that this nematode may reduce yields by 80% in chickpea and wheat (Di Vito and Greco, 1988b).

Known Hosts

Investigations on the host range of this root-knot nematode indicated that *M. artiellia* reproduces well on cereals, cruciferae, and leguminosae.

Apium graveolens (celery), *Artemisia* spp. (sagebrush), *Avena sativa* (oat), *Beta vulgaris* ssp. *vulgaris* (mangel), *Brassica napus* var. *napobrassica* (rutabaga), *Brassica oleracea* (cabbage, broccoli, kale), *Brassica rapa* (turnip), *Brassica* spp., *Cicer arientinum* (chickpea), *Hedysarum coronarium* (sulla), *Hordeum vulgare* (barley), *Lathyrus cicera* (lesser pea), *Lathyrus sativus* (grass pea), *Lens* spp. (lentils), *Medicago sativa* (alfalfa), *Medicago* spp. (medic), *Nasturtium fontanum* (rashad), *Phaseolus vulgaris* (bean), *Pisum sativum* (pea), *Raphanus sativus* (radish), *Sorghum vulgare* (sorghum), *Trifolium incarnatum* (crimsom clover), *Trifolium pretense* (red clover), *Trifolium repens* (white clover), *Triticum aestivum* (wheat), *Triticum durum* (durum wheat), *Triticum* spp. (wheat), *Triticum vulgare* (common wheat), *Vicia faba* (broad bean), *Vicia monantha* (garden vetch), *Vicia sativa* (vetch), and *Vicia* spp. (vetch) (CABI, 2004; Davis and Venette, 2004).

Known Vectors (or associated organisms)

In two genotypes of chickpea with complete resistance to Fusarium wilt, infection by *M. artiellia* overcame the resistance to *Fusarium oxysporum* f. sp. *ciceris* race 5 (Castillo et al., 2003).

Known Distribution

The British root-knot nematode occurs in northern Europe, the Mediterranean, North Africa, the Middle East, Russia, and China.

Africa: Algeria, Morocco, and Tunisia. **Asia:** China, Israel, Russia, Syria, and Turkey. **Europe:** United Kingdom, France, Greece, Italy, and Spain.

Potential Distribution Within the United States

As with other plant pests occurring in the Middle East, there is risk of introducing *M. artiellia* into the United States because of the intensification in the movement of machinery and personnel between the United States and Middle Eastern (Greco et al., 1992).

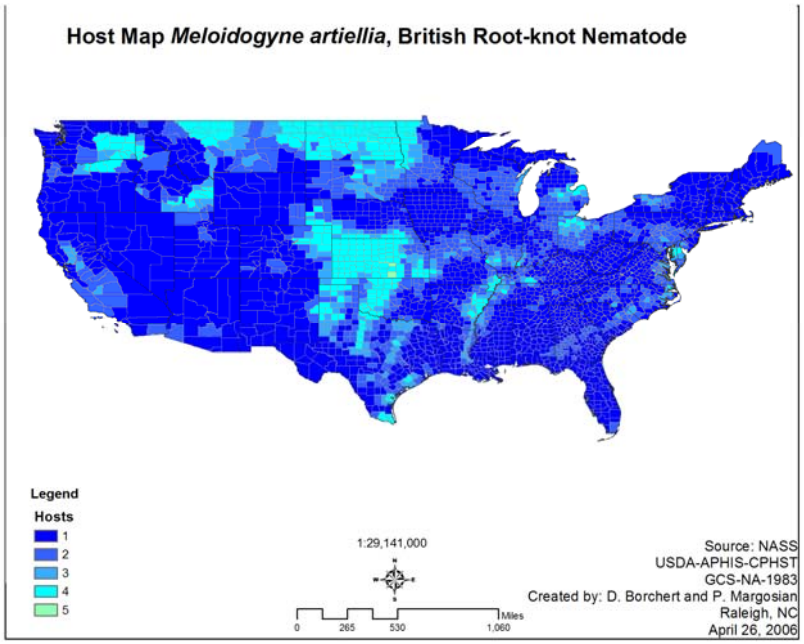

Figure 4. Host risk map for *M. artiellia* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Little information is currently available on the potential distribution within the United States, but areas that plant mustard crops, legumes, and small grains are at risk from introductions. At this time, a host risk map is available (Fig. 4). The host risk map describes the relative density (on a scale of 1-10) of susceptible hosts. The maps are based on National Agricultural Statistics Service (NASS) data. The scale of one to ten describes the proportion of total host acreage per county: for example a rank of one indicates no host acreage, while a score of ten indicates that 75-100% of the acres in the county contain suitable hosts for the pest. This map shows that Sumner and Harvey Counties in Kansas have the greatest risk of *M. artiellia* (rating of 5) based on host availability. Portions of Arkansas, Colorado, Deleaware, Idaho, Illinois, Indiana, Kansas, Kentucky, Maryland, Michigan, Missouri, Montana, Nebraska, New Mexico, North Dakota, Oklahoma, Ohio, Oregon, South Dakota, Tennessee, Texas, and Washington where small grains are grown are at risk (ranking of 4) from this nematode based on host availability.

Survey

Preferred Method:

Leguminous crops (clover and vetch) and crucifers (cabbage, cauliflower, radish, and turnip) are checked during cool months for patches of stunted plants with chlorotic leaves or other nutrient deficiency symptoms. Soil and roots from these plants are collected and the roots examined with the aid of a stereomicroscope for the presence of galls and nematode egg masses adhering to the small galls. Note: The standard procedure of egg mass staining with ploxine B cannot be used for *M. artiellia* detection because the egg masses of this nematode do not retain the stain (Greco et al., 1992).

Vovlas and Inserra (1996) outline general considerations for conducting a survey for a new *Meloidogyne* spp in citrus orchards. In general, they recommend sampling root tissues to inspect for the presence of galled roots. They also note that soil samples may detect *Meloidogyne* spp., but these individuals may not be of particular concern. Many native or naturalized *Meloidogyne* spp. parasitize a number of weed hosts. Thus, careful examination of individuals will be necessary to confirm species identity. Samples of soil or host roots must be collected with the purpose of obtaining males, juveniles, or nematodes within root tissues. Samples must then be processed to separate nematodes from soil and debris. Finally, nematodes must be prepared either for identification using morphological (e.g., perineal patterns) or molecular techniques.

Root-knot nematodes are extracted from soil using a variety of techniques. Six methods (and subtle variations thereof) are particularly common: Baermann trays; Baermann trays with elutriation or sieving; centrifugal flotation; flotation-sieving; semiautomatic elutriation; and Cobb's decanting and sieving. These methods are described in detail by Barker (1985). The efficiency of the nematode extraction is influenced by the amount of soil that is processed at one time. Extraction efficiencies are greatest when 100 g to 450 g of soil are processed. Extraction efficiencies for *Meloidogyne* spp. are frequently low and can vary between 13 and 45% (Davis and Venette, 2004).

Key Diagnostics

M. artiellia may occur in mixed populations with closely related or other easily confused species. Swollen females and egg masses of *M. artiellia* are distinctly large but can be easily confused with other root-knot nematodes without close examination using magnification. Due to technological advances in identification techniques, differentiating among morphologically similar root-knot nematodes can be accomplished most reliably by restriction fragment length polymorphism (PCR-RFLP) of ribosomal DNA (DeGiorgi et al, 1991, 1994, 2002; Davis and Venette, 2004).

Easily Confused Pests

Meloidogyne artiellia can be confused with other root-knot nematodes. *Meloidogyne artiellia* can be easily distinguished morphologically from other root-knot nematodes reported in the United States as follows: *M. artiellia* second-stage juveniles (J2s) have tail 18-26 µm long, whereas the J2 of *M. acrita*, *M. arenaria, M. christiei*, *M. cruciana*, *M. megatyla*, *M. hapla*, *M. incognita*, *M. graminis*, *M. javanica*, *M. querciana*, and *M. thamesi* have tail length of >30 µm. Body length parameters of *M. artiellia* J3 range 300-370 µm and overlap with those of *M. acrita* (345-396 µm), *M. incognita* (360-393 µm), and *M. javanica* (340-400 µm), but they are smaller than those (>370 µm) of other root knot nematodes reported in the United States (Greco et al., 1999). Female *M. artiellia* also have a cuticular perineal pattern with a very distinct inner area containing the vulva and anus. This area is marked by a few coarse striae in an eight-shaped figure with a large base and a small top.

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Secondary Pests of Small Grains (Truncated Pest Datasheet)
None at this time

Tertiary Pests of Small Grains (Name and Photo only)
None at this time

Plant Pathogens

Primary Pests of Small Grains (Full Pest Datasheet)

Puccinia graminis* f. sp. *tritici race Ug99 - North American race designation: TTKS

Scientific Name
Puccinia graminis f. sp. *tritici* Ug99 Eriks. & E. Henn.

Synonyms:
Puccinia graminis var. graminis, Puccinia graminis f.sp. avenae, Puccinia graminis subsp. graminis, Puccinia graminis subsp. graminicola

Common Name(s)
Stem rust of cereals, wheat rust, stem rust, black stem rust, black stem rust of cereals, black rust, black rust of cereals, wheat stem rust, barley stem rust

Type of Pest
Fungus

Taxonomic Position
Phylum: Basidiomycota, **Class:** Basidiomycetes, **Order:** Uredinales, **Family:** *Puccinia*ceae

Reason for Inclusion in Manual
National Threat

Pest Description
Puccinia graminis f. sp. *tritici* race Ug99 is a new virulent physiological race of wheat stem rust, which was discovered in Uganda in 1999. The new race was later found in Kenya in 2001 and in Ethiopia in 2003. The rust has now spread from eastern Africa across the Arabian Peninsula and is infecting wheat in Yemen, Sudan, and Iran. Given that the spores of the fungus can travel great distances on the wind and on the clothing of airline passengers, the new race

could spread to the vast wheat-growing areas of North Africa, the Middle East, Pakistan, India, and Southeast Asia, where it has potential to cause major crop losses. Wheat grown in other continents (e.g., North America) is also at a great risk from Ug99, as studies have shown that many of the wheat cultivars currently grown, in addition to barley, are susceptible to this new race (NAPPO, 2007).

Puccinia graminis is a macrocyclic rust, with five distinct spore stages and requires the alternate host of barberry for sexual reproduction.

Uredinial Stage: The uredinial, or red summer, stage is initiated by germination of a urediniospore on its grassy host, penetration, development of an intracellular mycelium with intracellular haustoria, and subsequent sporulation of uredinia to form new urediniospores. The recycling of the uredinial stage is the major means whereby the fungus initiates and perpetuates an epidemic. The urediniospores of *P. graminis* are dikaryotic (n+n), dehiscent, thick-walled and covered with spines. They are elliptical and about 20 x 30 µm.

Telial Stage: As infected plants mature, urediniospore formation ceases and teliospore formation commences, either in the same, or in new (telia), fruiting structures. At this stage, the infections become black, hence the name black rust. The ontogeny of teliospores is the same as urediniospores, but the teliospores remain attached. The teliospores are two-celled, thick-walled (with up to five wall

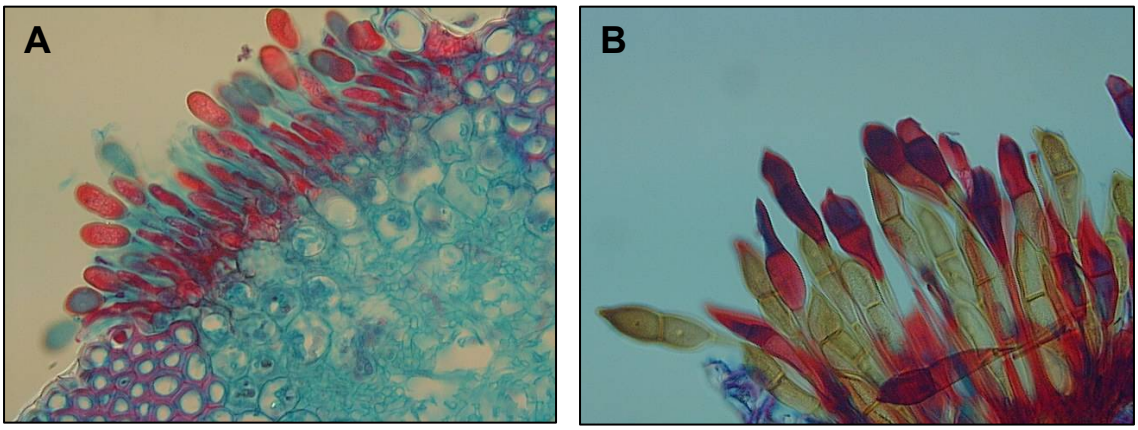

Figure 1. Urediniospores in uredia (A) and two-celled teliopsores in telia (B). Photos courtesy of R. Hammerschmidt.

layers) and are thickened at the apical end. Teliospores are important because they are constitutionally dormant, enabling the fungus to survive severe cold or drought. The mature teliospore represents the only true diploid state of the fungus.

Basidiospore Stage: The germination of teliospores and subsequent meiosis in the basidium results in the formation of haploid basidiospores. Four basidiospores, two of each opposite mating types, are produced from each basidium. If basidiospores are deposited on the surface of the alternate host,

usually *Berberis vulgaris* (barberry), they germinate, penetrate directly through the host epidermis and form a haploid mycelium. The fungus is most capable of infecting *Berberis* only when the leaves are young and tender. The fruiting structure, formed as a result of basidiospore infection, is called a pycnium.

Figure 2. *P. graminis* f. sp. *tritici* uredinia on wheat. Photo courtesy Yue Jin, USDA-ARS Cereal Disease Laboratory

Spermatial Stage: The pycnia are normally formed on the adaxial leaf surface, often in clusters. The important features of the pycnia are the formation of flexuous (receptive) hyphae and haploid spermatia. The spermatia, produced successively from the terminal ends of sporophores, are exuded in nectar. The nectar attracts insects, which in addition to splashing rain drops, serve to transport the spermatia to flexuous hyphae of the pycnia of opposite mating types, where fusion occurs.

Aecial Stage: Following union of the opposite mating types, dikaryotization occurs. The spermatial nuclei migrate to the protoaecium, where mitosis occurs, the nuclei reassort into dikaryons and the aecial structure forms. The aecia of *P. graminis* are elongated, cylindrical structures. The ornamented, dikaryotic aeciospores are produced successively in chains from the aeciosporophores. The aeciospores infect the grassy host, completing the fungal life cycle.

For additional information on the five spore stages of *P. graminis* f.sp. *tritici* see http://www.botany.hawaii.edu/faculty/wong/Bot201/Basidiomycota/Uredinomycetes/Uredinomycetes.htm.

Biology and Ecology

It is only in the most extreme climatic regions, such as very hot and dry, or tropical humid, that *P. graminis* does not occur, although irrigation can result in the occurrence of stem rust in some of these regions.

Although stem rust on wheat has largely been controlled worldwide by the use of resistant cultivars, The new stem rust race Ug99 is particularly concerning, however as it has broken historically affective resistance sources. Stem rust is potentially a continuous problem in some areas. Most regions of the world have conditions that are normally more marginal for rust development.

Figure 3. Aecial stage on barberry. Photo courtesy of A. J. Silverside.

Appropriate conditions such as source and timing of inoculum, susceptible hosts, temperature and humidity are required for rust development. When these conditions occur, disease losses may become severe. The distribution of the pathogen is affected by prevailing climatic conditions, the movement of global air masses, geographical features, the availability of alternative grassy hosts or the alternate sexual host, and cropping practices.
Urediniospores may be transported by wind over long distances, thus the occurrence of stem rust is only limited by the deposition pattern of the spores, suitable weather conditions and the availability of susceptible hosts. Geographic distribution must, therefore, be considered in terms of disease epidemiology (CABI, 2004).

Puccinia graminis f. sp. *tritici* is not seedborne.

Symptoms/Signs

The uredinia may occur on leaves, stems, leaf sheaths, spikes, glumes, awns and occasionally on grains of their grassy hosts; stems and leaf sheaths are the main tissues affected. On stems, the uredinia are elongated and reddish-brown (Fig. 2); loose epidermal tissue is conspicuous at the margins of the uredinia, giving a roughened feel to the stem surface. The uredinia coalesce to cover large areas of the host tissue in heavy infection. Since the urediniospores are dehiscent, they are released as powdery masses from the uredinia.

The telial stage occurs in the same tissue as the uredinial stage, but becomes shiny-black. The teliospores are sessile, and the telial tissue is, therefore, firmer

than the uredinial tissue; no spores are released.

The pycnial stage occurs on the young leaves of the alternate host, mainly *Berberis vulgaris*. Pycnial infections initially appear as light, chlorotic areas on the adaxial leaf surface, then become light orange-brown lesions, consisting of individual small cone-shaped eruptions (the pycnia), often occurring in clusters.

The aecia develop on the abaxial surfaces of the leaves of the alternate host. When mature, they appear as bright-orange, closely-packed, raised clusters of individual aecia (Fig. 3). The aecia are cylindrical in shape and flare out at their apices, appearing as a grouping of rings within the aecial cluster.

Pest Importance

Prior to the widespread use of modern varieties with resistance to *P. graminis* f.sp. *tritici,* several epidemics resulted in massive losses in North America during the early part of the 20th century (Expert Panel on the Stem Rust, 2005). One such epidemic resulted in the loss of 200 million bushels of wheat (Partridge, 2000).

Stem rust remains a potential problem on cultivated cereals, mainly in the northern plains region of the United States (Minnesota, North Dakota and South Dakota) and the eastern prairie region of Canada (Manitoba and eastern Saskatchewan). Historically, the occurrence of stem rust may have been more widespread. *Berberis,* the alternate host, eradication laws were passed in Massachusetts as early as 1754, followed soon thereafter by Rhode Island and Connecticut. *Berberis vulgaris* has now been eliminated from most of the United States and Canadian small grain cereal growing area (Roelfs, 1982).

The recent development of a new race of *P. graminis* (Ug99) that is virulent of wheat lines with the widely used stem rust resistance gene *sr31* represents a threat to wheat producing regions worldwide (Jin and Singh, 2006; Singh et al., 2006; Wanyera et al., 2006). This race also has virulence to genes *sr5, 6*, *7b, 8a, 8b, 9a, 9b, 9d, 9e, 9g, 11, 15, 17, 21, 30,* and *38* (Expert Panel on the Stem Rust, 2005). Annual losses of as much as 3 billion in Africa, the Middle East, and South Asia alone are possible.

Known Hosts (*P. graminis*)

Major hosts

Avena sativa (oats), *Hordeum vulgare* (barley), *Secale cereale* (rye), *Triticum aestivum* (wheat), *Triticum turgidum* (durum wheat)

Minor hosts

turfgrasses

Wild hosts

Berberis vulgaris (European barberry), *Lolium multiflorum* (Italian ryegrass)

Known Vectors (or associated organisms)

Puccinia graminis f.sp. *tritici* race Ug99 is not a known vector and does not have any associated organisms.

Known Distribution

Puccinia graminis f. sp. *tritici* has a worldwide distribution on cultivated cereals and grassy hosts. The Ug99 strain has been reported from Uganda, Kenya, Ethiopia, Sudan, Yemen, and most recently Iran. A recent Offshore Pest Information System (OPIS) report, dated 3-13-08, indicates that this pathogen is present in Iran.

Potential Distribution within the United States

Ug99 will eventually reach North America. The most likely avenues will be air currents and passenger clothing transport of Ug99 spores from areas of the eastern hemisphere where the rust is present. Because the Ug99 strain of *P. graminis* f.sp. *tritici* is able to overcome the most commonly used source of resistance and infect previously resistant varieties, new epidemics are predicted to occur when the Ug99 strain enters the United States (NPAG, 2005). Eighty percent of the hard red spring wheat grown in the northern Great Plains has no resistance to Ug99 (ARS, 2006). All wheat production, and possible barley, production areas are at considerable risk from this new race of *P. graminis* f. sp. *tritici*.

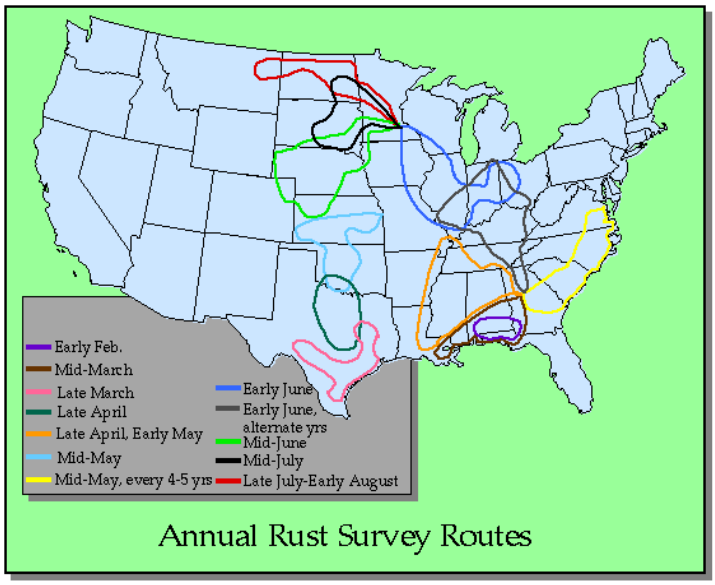

Figure 4. Survey routes used by the Cereal Disease Laboratory. Photo from http://www.ars.usda.gov/SP2UserFiles/Place/00000000/opmp/wheat%20rust%2008-28-06.pdf

Survey

Grain fields or forages can be inspected for the disease at any stage of growth. From the seedling stage until about the five- or six-leaf stage, stem rust infections are most obvious on the leaves. The main time for inspection is from the stage when the crop begins to head (late boot stage) until near maturity. Stem rust is generally detected in late April in the southern United States; much later than either leaf or strip rust. In July, stem rust is generally detectable on trap plots of susceptible winter and spring wheat, barley, and wild grasses in the northern Great Plains (Kolmer, 2007).

Stem rust pustules are easily detected by sweeping aside the canopy of growth and looking into the crop with strong sunlight coming from behind.

The red-brown pustules are easily recognized against the normally blue-green color of healthy stem tissue. However, there may be other causes for similar discoloration. Infections due to stem rust can usually be differentiated from these other causes by gently feeling the plant tissue between the thumb and forefinger for the roughened surface caused by rust infection. This test may also be used for detached plant tissue, where a rough feel of red-brown or black lesions may indicate the presence of rust fungal uredinia or telia, respectively (CABI, 2004).

The USDA Cereal Disease Laboratory at the University of Minnesota conducts annual surveys of wheat, oat, barley, and rye stem rust, as well as wheat leaf rust and oat crown rust. Each year, scientists make seven survey trips (Fig. 4) through representative areas of the Great Plains and the Midwestern states from Missouri to Ohio to monitor rust developments in small grain crops and to collect rust samples. From the samples, they identify the rust races present in the infected crop by inoculating small grain varieties with different specific resistance genes. The identification of rust races and the collection of knowledge regarding rust resistance genes is a national service provided by this laboratory.

Key Diagnostics

The diagnosis of stem rust infections is principally made by means of the fungal signs. A scraping of the lesion(s) may be taken with a scalpel, transferred to a glass microscopic slide and placed in a droplet of water. The slide can be examined under low magnification (100 X) for urediniospores. The urediniospores of *P. graminis* generally appear as oblong, spiney spores, about 20 x 30 µm, of a light-orange to tan color. The host species normally indicates the *formae specialis* of the pathogen, particularly in a cultivated host; barley is an exception (CABI, 2004). A set of host differentials with different resistant genes will need to be inoculated to identify whether the Ug99 race is present.

Real time PCR protocols have been reported to detect and identify *P. graminis, P. striiformis, P. recondita* and *P. triticina* (Barnes and Szabo, 2007) at the genus and species level but not at the race level. For *Puccinia graminis* f.sp. *tritici* race Ug99, host differentials will have to be inoculated to determine the pathogen race.

Easily Confused Pests

It is possible to confuse *P. graminis* f. sp. *tritici* race Ug99 with other *Puccinia* spp., other *formae specialis* of *P. graminis*, and other races of *P. graminis* f. sp. *tritici*. Molecular tests, host range, and inoculation of differentials to determine race may be necessary to accurately identify *P. graminis* f. sp. *tritici* race Ug99.

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Rice Hoja Blanca virus

Scientific Name
Rice hoja blanca tenuivirus

Synonyms:
RHBV, hoja blanca, rice white leaf virus,

Common Name(s)
Hoja blanca, white leaf disease of rice, chlorosis of rice, rice white leaf virus, hoja blanca of rice

Type of Pest
Plant pathogenic virus

Taxonomic Position
Unassigned virus family: Tenuivirus

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description
Rice hoja blanca virus (RHBV) is a member of the tenuivirus group. The virus particles are 10 nm wide and up to 3 µm long and spiral, filamentous particles up to 3 nm in diameter have been reported. Four single-stranded RNA (ssRNA) strands of plus and minus polarities and coat protein (Morales and Niessen, 1983; Espinoza et al., 1992; Hibino, 1996) are possible. The total genome is estimated to be 17.6 kb. The RNA 1 is 9.8 kb and encodes for a RNA dependent RNA polymerase on the negative strand. The RNA 2 is 3620 nucleotides and RNA 2 is 2299 nucleotides, both of which encode for two proteins using an ambisense coding strategy. The nucleoprotein gene (N) is encoded by the virus complementary strand of RNA 3. The RNA 4 consists of 1991 nucleotides and encodes for two proteins using an ambisense coding strategy (Calvert et al., 2003). RHBV is efficiently transmitted by the planthopper, *Tagosodes orizicolus*, but is not mechanically or seed transmittable (CABI, 2004).

Symptoms/Signs
RHBV-infected rice plants show stunting, chlorotic or yellow stipling (stripes) and mottling of leaves (Fig. 1), and premature wilting (Hibino, 1996). In some cases, the whole leaf blade may turn white. Sterility and death of the plant normally follow. The palea and lemma are distorted and turn brown. Infected plants also have fewer and smaller roots, and the roots may become necrotic. Symptoms develop from 5 to 34 days after infection, depending on the age of the plant, the cultivar, and the site of infection (Hibino, 1992).

Wheat, rye, barley, oats, and some grasses are also hosts for this virus. Wheat becomes infected especially when grown adjacent to rice and shows a characteristic and prominent gray-white discoloration of the spike and uppermost leaves, sometimes referred to as “white tip” or “white spike” (Weiss, 1998).

Figure 1. *Rice hoja blanca virus* in rice, yellowing, chlorosis of leaves Photo courtesy R.K. Webster and P.S. Gunnell.

Pest Importance

Hoja blanca occurs in Central and South America and the Caribbean and is the only recognized virus disease of rice on the American continents. It was observed in Colombia as early as 1935 but did not become an economically important disease until 1956-1965, when it was responsible for up to 80% yield loss in several countries (Hibino, 1992, 1998; Morales and Niessen, 1983). From 1967 to 1980, the disease incidence was low, but during 1981-1985 the disease because a problem again, especially in Central America. The cyclical appearance of the disease may be explained by the deleterious effect of RHBV on the vector population (Hibino, 1992).

Known Hosts

Major hosts

Avena sativa (oats), *Oryza sativa* (rice), *Triticum* spp. (wheat)

Minor hosts

Hordeum vulgare (barley), *Secale cereale* (rye)

Known Vectors (or associated organisms)

RHBV is transmitted in a persistent manner by the planthopper *Tagosodes orizicolus (*synonym *Sogatodes oryzicolus)* (Hibino, 1996). RHBV is propagative in the vectors and is transmitted at a high rate from female adults to their progeny via eggs. There is a high rate of transovarial transmission to the progeny, and these nymphs can transmit the virus shortly (approximately 30 minutes) after

they emerge (Galvez et al., 1968). *T. orizicolus* with newly acquired RHBV becomes infective after an incubation period of 30-36 days. Because of the long incubation period of the virus, *T. orizicolus* planthoppers that acquire RHBV through feeding on infected plants rarely live long enough to transmit the virus. *T. orizicolus* infected with RHBV have a shortened life span, low fecundity, and reduced nymph viability (Hibino, 1996). The planthoppers that acquire the virus via transovarial transmission are, therefore, thought to be the primary vectors of this virus (Hibino, 1992).

Figure 2. Panicles of infected tillers may be completely sterile, with malformed and discolored florets and grains. Photo courtesy of R.S. Zeigler (CABI, 2004).

Under controlled conditions, hoja blanca infection predisposes rice plants to brown spot caused by *Cochliobolus miyabeanus* (Lamey and Everett, 1967).

Known Distribution

Caribbean: Cuba, Dominican Republic, Puerto Rico, Trinidad and Tobago. **Central America:** Belize, Costa Rica, El Salvador, Guatemala, Honduras, Nicaragua, Panama. **North America:** Mexico. **South America:** Argentina, Brazil, Colombia, Ecuador, Guyana, Peru, Suriname, Venezuela

Potential Distribution within the United States

In the United States, the insect vector and/or the disease were observed sporadically from 1957 to 1959 and 1962 in Florida, Louisiana, and Mississippi (CABI, 2004). Programs established to eradicate the planthopper vector and the virus were successful, and the disease has not reoccurred (Hibino, 1992). Wheat grown apart from rice or in association with resistant rice cultivars is rarely infected (Weiss, 1998). Therefore, areas that grow susceptible rice cultivars close to wheat are the most susceptible. Areas where both rice and wheat are grown include portions of Arkansas, California, Louisiana, Missouri, and Texas (Fig. 3).

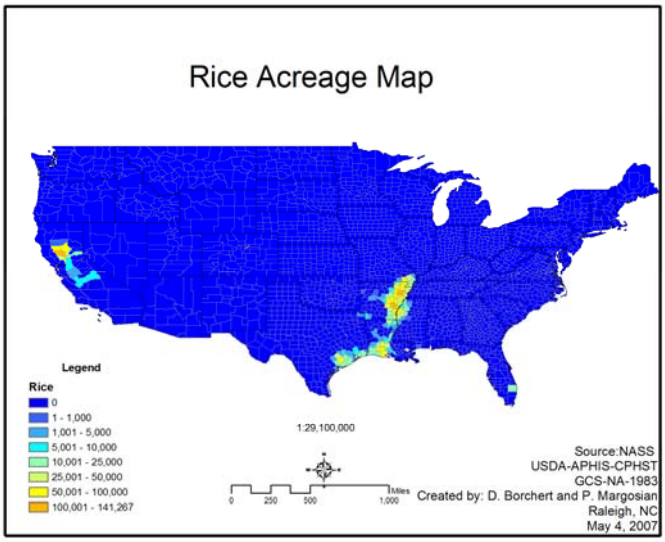
Survey

RHBV-infected rice plants show stunting, chlorotic or yellow stipling (stripes) and mottling of leaves, and premature wilting. In some cases, the whole leaf blade may turn white. Serology (ELISA) is then used to confirm the diagnosis.

Key Diagnostics

Rice cells infected with RHBV contain large masses of fine filaments, 8-10 nm in width in the nuclei and cytoplasm. However; this is only visible using electron microscopy. Infected plants produce a large amount of virus-specific proteins.

Rice plants infected by RHBV can be recognized by the white leaf symptom. Serology is used to confirm the diagnosis, and an ELISA (Falk et al., 1987) has been developed for the detection of both the nucleoprotein and the major NS4 protein. Using ELISA, the virus can be detected in infected planthoppers and plants (Zeigler and Morales, 1990).



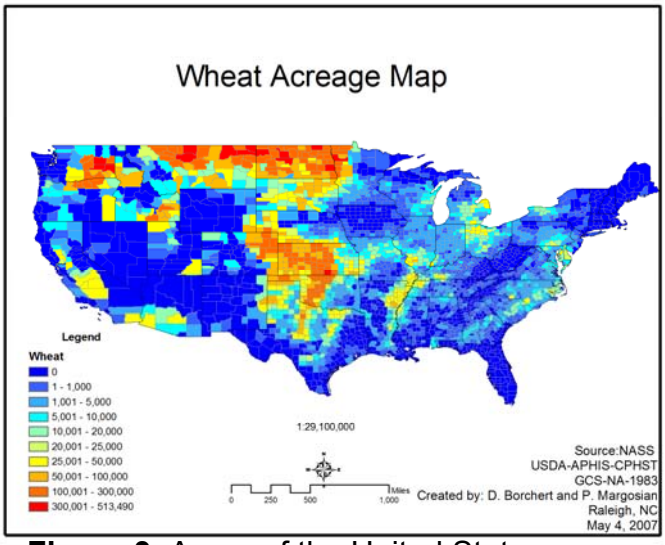


Figure 3. Areas of the United States were rice and wheat are grown. Note: there is overlap in Arkansas, California, Louisiana, Missouri, and Texas.

Easily Confused Pests

Rice hoja blanca virus can be distinguished from other rice viruses by differences in particle morphology, antigenic and vector specificity, and/or symptomatology. Rice stripe virus, which induces similar striping symptoms, causes malformation in the newly developed leaves; whereas the new leaves of hoja blanca-affected plants unfold normally (Morales and Niessen, 1985).

RHBV is serologically related to the Echinochloa hoja blanca virus (EHBV), which produces symptoms like RHBV on the weed host *Echinocloa colunum* and is

transmitted by the vector *Tagosodes cubanus*. EHBV may be a strain of RHBV (Morales and Niessen, 1985). ELISA does not distinguish between RHBV and EHBV.

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Peronosclerospora philippinensis

Scientific Name
Peronosclerospora philippinensis (W. Weston) C.G. Shaw

Synonyms:
Pernosclerospora sacchari, Sclerospora indica, Sclerospora maydis, Sclerospora philippinensis

Common Name
Philippine downy mildew, Java downy mildew of corn, sugarcane downy mildew

Type of Pest
Fungus-like

Taxonomic Position
Phylum: Oomycota, **Class:** Oomycetes, **Order:** Peronosporales, **Family:** Peronosporaceae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe; Agricultural Bioterrorism Protection Act of 2002; Regulated Plant Pest List

Pest Description
Philippine downy mildew is confined to parts of Asia and has not been reported within the United States. This obligate pathogen causes a serious disease to two major crops, corn and sugarcane. Yield losses of 40-60% and 25% have been observed in corn and sugarcane respectively (USDA, 1986). Oat (*Avena* spp.) is considered a minor host for Philippine downy mildew.

Figure 1. Chlorotic symptoms of Philippine downy mildew. Photo courtesy of CIMMYT.

The mycelia are branched, slender (8 µm in diameter), irregularly constricted and inflated. The small haustoria (2 x 8 µm) are simple and vesiculiform to subdigitate. Erect conidiophores (15-26 x 150-400 µm) grow out of stomata and are dichotomously branched two to four times. Branches are

robust. Sterigmata are ovoid to subulate, slightly curved, and 10 µm long. The conidia (17-21 x 27-39 µm) are elongate ovoid to round cylindrical, hyaline, and slightly rounded at the apex (Weston, 1920; White, 1999).

Oospores are rarely produced and are not produced in corn tissue (White, 1999).

Symptoms/Signs

Symptoms of the disease and signs of the pathogen are not listed specifically for oat.

On corn, the first symptoms typically appear as chlorotic stripes at the first leaves as early as 9 days after planting. All leaves on a plant may show characteristic symptoms of long chlorotic (yellow) streaks (Fig. 1). Weston (1920) reported the collapse of badly infected cells and the destruction of chloroplasts, resulting in the characteristic yellow color of diseased leaves.

A downy (grayish) covering primarily on the underside of the leaves is characteristic beginning at the two-leaf stage and is present until the appearance of tassels and silks. This covering is the site of spore production (conidia on conidiophores) and the source for secondary spread of the disease to other susceptible plants. As the plant ages, leaves may narrow, become abnormally erect, and appear somewhat dried-out. As the corn plant matures, tassels become malformed and produce less pollen, ear formation is interrupted, and sterility of seeds can result (Expert Panel, 2006). If infection occurs early, plants are stunted and may die (White, 1999).

There are no external symptoms on seeds. The fungus becomes established in the pericarp layer in the form of mycelium. The fungus is also present in the embryo and endosperm. There are no reports on the effect of *P. philippinensis* on seed quality (CABI, 2004).

Figure 2. Downy growth characteristic of downy mildews. Photo of downy mildew of tobacco. Photo courtesy of Tom Creswell, North Carolina State University. www.invasive.org

Note: There is a synergistic relationship between downy mildew fungi and maize streak

virus (MSV) on corn. Infection by MSV can mask symptoms of downy mildew infection. Reduction in height and biomass were significantly greater with pathogen combinations than with single pathogens (Damsteegt et al., 1993).

Survey

Survey is conducted via visual survey of plants for symptoms of the disease (chlorotic stripes or streaks) and signs of the pathogen (downy growth on underside of leaves consisting of conidia and conidiophores). Spore trapping using Burkhard spore traps and sentinel plots (unsprayed, susceptible plants that are scouted regularly) are suggested for early detection and have been employed for resistance screening (Cardwell et al., 1997; Expert Panel, 2006).

Key Diagnostics

Peronosclerospora spp. and other downy mildew genera (including *Sclerospora* and *Sclerophthora*) are primarily differentiated by pathogen morphology, including condiophore structure and dimension and spore (conidia) shape and size. However, these characteristics can vary considerably under different culture conditions, at different developmental stages, and on different hosts. Characters for downy mildew pathogens are listed in Table 1. Isozyme comparisons have been used to identify *Peronosclerospora spp.*, including *P. philippensis* (Bonde et al., 1984; Micales et al., 1988). *P. philippinensis* is indistinguishable from *P. sacchari* by isozyme analysis and many experts have synomized the two species. Some DNA-based approaches have been reported for other *Peronosclerospora.* Yao et al. (1991a) created a DNA clone that could be used as a species-specific hybridization probe for the detection and identification of *P. sorghi*. Yao et al. (1991b) then developed a DNA probe from a *P. maydis* genomic library to detect *P. sacchari* in maize leaves and seeds.

Table 1: Characteristics of the downy mildew fungi found on corn and other hosts
Taken from White (1999)

PATHOGEN	***PERONOSCLEROSPORA***				***SCLEROSPORA***	***SCLEROPTHORA***	
	philippinensis (Philippine downy mildew)	*maydis* (Java downy mildew)	*sacchari* (sugarcane downy mildew)	*sorghi* (sorghum downy mildew)	*graminicola* (Graminicola downy mildew)	*macrospora* (crazy top)	*rayssiae var. zeae* (brown stripe downy mildew)
Conidiophores	Hyaline, length 150-400 µm, bloated, widening abruptly, dichotomously branched 2-4 times, ephemeral	Hyaline, length 150-550 µm, bloated, dichotomously branched 2-4 times, ephemeral	Hyaline, length 160-170 µm, bloated, widening gradually, dichotomously branched 2-3 times, ephemeral	Hyaline, length 180-300 µm, bloated, often dichotomously branched 2-3 times, septate near base, ephemeral	Hyaline, length av. 268 µm, bloated, non-septate, irregularly dichotomously branched ephemeral	Hyaline, length av. 13.8 µm, simple, hyphoid, determinate	Hyaline, short, hyphoid, determinate
Asexual spores	Conidia hyaline, elongate-ovoid to round-cylindrical, apex slightly rounded, 17-21 x 27-39 µm	Conidia hyaline, spherical to subspherical, 17-23 x 27-39 µm	Conidia hyaline, elliptical, oblong or conical, apex round, 15-239 x 25-41 µm	Conidia hyaline, oval to almost spherical, 15-26.9 x 15-28.9 µm	Sporangia hyaline, broadly elliptical, operculate, papillate, 12-21 x 14-31 µm	Sporangia hyaline, lemon-shaped, operculate, papillate, 30-60 x 60-100 µm	Sporangia hyaline, ovate to almost cylindrical, operculate, 18.5-26 x 29-66.5 µm
Asexual spores germinate by	Germ tube	Germ tube	Germ tube	Germ tube	Zoospores	Many zoospores	Zoospores

PATHOGEN	***PERONOSCLEROSPORA***				***SCLEROSPORA***	***SCLEROPTHORA***	
	philippinensis (Philippine downy mildew)	*maydis* (Java downy mildew)	*sacchari* (sugarcane downy mildew)	*sorghi* (sorghum downy mildew)	*graminicola* (Graminicola downy mildew)	*macrospora* (crazy top)	*rayssiae var. zeae* (brown stripe downy mildew)
Oospores	Rare or nonexistent; spherical, smooth walled, 22.6 µm in diameter	Unknown	Yellow to yellow brown, globular to slightly angular; 40-50 µm in diameter	Usually brown to subhyaline spherical, diameter 25-42.9 µm	Pale brown, spherical, usually smooth-walled, diameter 22.5-35 µm	Hyaline to pale yellow, mainly in vascular bundles, diameter 45-75 µm	Brown, spherical, 29.5-37 µm in diameter
Sexual spores germinate by	Side germ tube	-	Germ tube	Wide germ tube	Germ tube or by sporangia	Sporangia	Sporangia
Geographical Distribution	Philippines, Indonesia, India, Nepal, China, Thailand, Pakistan	Australia, Indonesia	Australia, Fiji Islands, Japan, Nepal, New Guinea, India, Philippines, Taiwan, Thailand	North America (including the U.S.), Central America, Asia, Africa, South America, Australia, Europe	Worldwide, but on corn found only in Israel and the United States	North America (including the U.S.), Mexico, South America, Europe, Africa, Asia	India, Nepal, Pakistan, Thailand

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Tertiary Pests of Small Grains (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to small grains and exotic to the US

None at this time

Weeds/Parasitic Plants

Primary Pests of Small Grains (Full Pest Datasheet)
None at this time

Secondary Pests of Small Grains (Truncated Pest Datasheet)

Arctotheca calendula

Scientific Name
Arctotheca calendula (L.) Levyns

Synonyms
Arctotheca calendulaceum, Arctotis calendula, Arctotis calendulacea, Artoctis calendula, Cryptostemma calendula, Cryptostemma calendulacea, Cryptostemma calendulaceum, Venidium decurrens

Common Name
Capeweed, cape marigold, Namaqualand daisy

Type of Pest
Weed

Taxonomic Position
Phylum: Magnoliophyta, **Class:** Magnoliopsida, **Order:** Asterales, **Family**: Asteraceae

Figure 1. Capeweed flower. Photo courtesy of Frank Hrusa. CDFA.

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe

Pest Description (from Cunningham et al., 1981)
Flowers/Seedhead: Many small flowers (florets) in solitary heads,

2-6 cm across (Fig. 1) the end of stalks 8-25 cm long. *A. calendula* flowers mostly in spring and early summer.

Description: An annual rosette-forming herb with taproot; individual plants to 80 cm wide and 30 cm high. Leaves with upper surface hairless to hairy; basal leaves 5-25 cm long, 206 cm wide, on a stalk to 6 cm long; upper leaves, if present, stem-clasping.

Distinguishing features: Distinguished by deeply lobed basal leaves, white downy underneath; area where seeds attach to the head (receptacle) pitted; petal-like ray florets yellow above, gray-green below with strip-like parts mostly 1.5-2.5 cm long, disc florets dark purple; seeds covered in pale brown wool and topped by 6-8 short scales.

Figure 2. Capeweed plants and infestation. Photos courtesy of Frank Hrusa. CDFA.

Arctotheca calendula is a native of South Africa and is currently present in California in coastal Marin and Humboldt counties. However, Hickman (1993) reports a probable range of coastal counties from the Oregon border to Monteray County. Herbicide resistance (paraquat) has been reported for *A. calendula* (Powles et al., 1989).

Symptoms/Signs

Capeweed competes with crop species and can have a detrimental effect on crop growth and yield. Capeweed has been found in wheat, barley, and oat fields. Additionally, capeweed has been shown to be a weed reservoir of *Thrips tabaci* and tomato spotted wilt virus in lettuce in Tasmania (Wlson, 1998).

Capeweed grows over and displaces other herbs. In coastal grasslands and riparian zones, it forms monospecific stands of impenetrable mats up to several thousand square feet (University of California, 2007). Capeweed is a rapidly growing groundcover. If planted on one-foot centers, capeweed will establish full cover within six months. According to McIvor and Smith (1973), capeweed has a higher relative growth rate in the early weeks of growth than clover and annual ryegrass. However, a marked reduction in growth was only shown if capeweed was established four weeks before clover.

Capeweed can accumulate potentially toxic levels of nitrate, and livestock deaths have been recorded following grazing of capeweed infested pastures (Fairnie, 1969). However, toxic levels of nitrate are only likely to be present in plants growing in high fertility soils, for example in stock yards. Milk from dairy cows feeding on the weed can have tainted milk. Horses and donkeys can have allergic skin reactions to the pollen encountered as they graze on the plant.

Survey

Preferred method: Survey for *A. calendula* involves visual survey. The optimum time for a survey is when capeweed is at peak flowering. Capeweed grows best in full sun to light shade, can tolerate a wide variety of soil types, and needs little water to persist once it is established.

Weed maps are typically produced from data sampled at discrete intervals on a regular grid. Errors are expected to occur as data are sampled at increasingly coarse scales. To demonstrate the potential effect of sampling strategy on the quality of weed maps, Cousens et al. (2001) analyzed a data set comprising the counts of capeweed in 225,000 quadrats completely covering a 0.9-ha area. The data were subsampled at different grid spacings, quadrat sizes, and starting points and were then used to produce maps by kriging. Spacings of 10 m were found to overestimate the geostatistical range by 100% and missed details apparently resulting from the spraying equipment. Some evidence was found supporting the rule of thumb that surveys should be conducted at a spacing of about half the scale of interest.

Lemerle et al. (1999) surveyed for weeds in canola, including *A. calendula*. At each field, weed species and plant density were recorded in twenty 1 m^2 quadrats. Five quadrats were spaced 50 m apart along each arm of a W-transect of the field. Crop growth stage, cultivar, and the soil type were recorded at each site. Voucher specimens were collected. The percentage incidence of a weed species was calculated using the mean of the 20 positions on the transect for each species over the 62 sites surveyed. The average weed plant density of each species was calculated for each site.

Key Diagnostics

Cypselar morphology provides a suite of taxonomic characters important at the genus and species levels in Arctotindinae, the subtribe to which *Arctotheca*

belongs. On the basis of morphology, *Arctotheca* is well defined and characterized by neuter ray florets, hermaphrodite disc florets, papillose filaments, bilaterally flattened cypselae with well-developed abaxial ridges but lacking wings, and a pappus never longer than the cypsela (McKenzie et al., 2005; McKenzie et al., 2006). McKenzie et al. (2005) provide a diagnostic key based on cypselar morphology. Cape marigold or African daisy, *Dimorphotheca sinuate*, is a winter annual with orange to yellow ray flowers, sometimes with violet tips or bases, and yellow disk flowers that has escaped cultivation and may be confused with capeweed. Unlike capeweed, cape marigold has phyllaries in 1 row, flowers that lack a pappus, smooth and tubercled achenes without woolly hairs, foliage covered with glandular hairs, and leaves that lack a dense covering of white woolly hairs (CDFA, 2007).

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Wilson, C.R. Incidence of weed reservoirs and vectors of tomato spotted wilt tospovirus on southern Tasmanian lettuce farms. Plant Pathology 47: 171-176.

Moraea spp.

Scientific Name
Moraea flaccida (Sweet) Steud.
Morea miniata Andrews

Synonyms:
Homeria flaccida, Homeria miniata

Note: Goldblatt (1998) transferred the genus *Homeria* and five additional genera to *Moraea* based on morphological, anatomical, and molecular analysis. *Moraea* currently includes over 200 species. This data sheet deals only with *Moraea flaccida* and *M. miniata*, which were formally *Homeria* spp.

Common Name
Cape tulip, one-leaf cape tulip, two-leaf cape tulip

Type of Pest
Weed

Taxonomic Position
Phylum: Magnoliophyta, **Class:** Liliopsida, **Order**: Liliales, **Family**: Iridaceae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe - (Listed as *Homeria* spp.)

Pest Description (From Australian Government, 2007)
Moraea flaccida and *miniata* are perennial weeds native to South Africa. In Australia, two-leaf cape tulip is less common than one-leaf cape tulip. The two species may grow together. Cape tulips can be a problem in oat fields.

Moraea flaccida: One-leaf cape tulip is a perennial herb with annual leaves and flowers to 60 cm high, developing from an underground bulb (corm) 1 to 4 cm in diameter,

Figure 1. Flowers of *M. flaccida* (top) and *M. miniata* (bottom). Photos courtesy of http://www.weeds.org.au/

which is white in color but covered in a brown fibrous tunic. Each plant has only one leaf which is flat, folded and ribbed, 1-2 cm wide and up to 1 m long (longer than the flowering stalk). The leaves are attached to the stem above ground level and often droop or trail on the ground. Flowers are borne on an erect, somewhat zig-zagged stem and are 3-5 cm in diameter. The six petals can vary from salmon-pink through orange to yellow in color. Each petal is 2.5 to 4 cm long. The fruit is a narrow, cylindrical, three-valved capsule up to 5.5 cm long which starts green and turns brown when mature, splitting from the apex to release the seeds. The brown seeds are irregular in shape and 1 to 2 mm long. Each capsule may contain up to 150 seeds.

Moraea miniata: Two-leaf cape tulip is a perennial herb with annual leaves and flowers to 60 cm high, developing from an underground bulb (corm) covered in a scaly, black tunic. There are usually clusters of cormils (small bulb-like structures) in the swollen leaf bases and often around the main corm (Fig. 2).

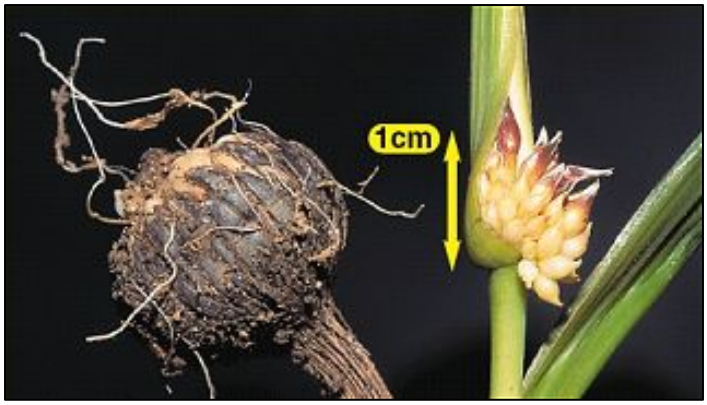

Figure 2. Small cormils form in the leaf axils (right) in *M. miniata*. Photo courtesy of http://www.weeds.org.au/

Each plant has 2-3 leaves which are flat, folded and ribbed, 1 to 2 cm wide and up to 100 cm long (longer than the flowering stalk). The leaves are attached to the stem above ground level and often droop or trail on the ground. Flowers are borne on an erect, somewhat zig-zagged stem. They are 2 to 4 cm in diameter with six pink petals with yellow bases dotted with green, each petal 1.3 to 2.5 cm long. Small capsules up to 1.5 cm long are sometimes formed after flowering, but these do not contain any seeds. Two-leaf cape tulip tends to grow in clumps if left undisturbed.

Cape tulips are difficult to control chemically due to the dormancy of corms below the ground. Cormil production may exceed many thousands per square meters, and corms may remain viable in the soil for many years.

Symptoms/Signs

Cape tulips compete with crop and native species and can have a detrimental effect on growth and yield. Both *M. flaccida* and *miniata* are highly toxic to livestock and may invade pastures and native vegetation. The poison is a glycoside which causes loss of appetite, weakness and depression, blindness, dysentery, scouring and paralysis of the hind legs. Death usually occurs within 3 days and treatment is charcoal or kaolin to absorb the poison. Livestock that are

accustomed to grazing on infestations are not affected as they know not to eat the plants (Murie, 1974; Australian Government, 2007).

Survey

Preferred method: Survey for *Moraea* spp. involves visual survey. The optimum time for a survey is when *Moraea* spp. is at peak flowering. Specific survey methods for cape tulip are lacking. However, weed maps are typically produced from data sampled at discrete intervals on a regular grid.

Key Diagnostics

One-leaf cape tulip can be confused with two-leaf cape tulip (*Moraea miniata*) and cape tulip. However, two-leaf cape tulip has 2 or 3 leaves, produces bulbils (small deciduous bulbs) in the leaf forks, does not produce seed, has a scaly covering around the corm and relatively broad petals (13 to 25 mm long). *Moraea ochroleuca* has yellow (rarely orange) flowers with relatively broad petals (30 to 40 mm long) (Navie 2004).

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Striga hermonthica

Scientific Name
Striga hermonthica (Del.) Benth.

Synonyms:
Buchnera hermontheca, Buchnera hermonthica, Striga hermontheca, *Striga senegalensis*

Common Name
Purple witchweed, giant witchweed

Type of Pest
Parasitic Plant

Taxonomic Position
Phylum: Magnoliophyta, **Class:** Magnoliopsida,
Order: Scrophulariales, **Family:** Scrophulariaceae

Reason for Inclusion in Manual
CAPS Target: CAPS Pest Universe, Federal Noxious Weed List

Pest Description
Striga hermonthica, a parasitic flowering plant, endemic to Africa, constitutes one of the most severe constraints to cereal production in sub-Saharan Africa. It parasitizes sorghum, maize, millet, rice and sugar cane, as well as pasture and wild grasses. *S. hermonthica* attaches itself to the roots of the host plant and diverts essential nutrients, which leaves the host stunted and yielding little or no grain. *S. hermonthica* often causes yield losses in excess of 50% (Parker, 1991). Barley is considered a minor host. There are only isolated reports of *Striga* spp. on wheat. Vasey et al. (2005) showed that wheat is highly susceptible to *S. hermonthica* under experimental conditions.

(From CABI, 2004)
Description: An herbaceous annual plant 30-100 cm high. *S. hermonthica* is obligately allogamous, requiring insect pollinators for fertilization and seed production (Musselmann, 1987). Larger plants may be branched. Stems and leaves are clothed in characteristic trichomes giving the plant a harsh texture. Leaves mostly opposite on the lower half of the stem but irregular above, narrowly lanceolate or elliptic, 2-8 cm long, up to 1 cm wide.

Flowers/Seedheads: Inflorescence a terminal spike of sessile flowers, with axillary spikes branching from upper leaf axils. Flowers subtended by bracts 1-2 cm long, up to 3 mm wide, with a fringe of ciliate hairs. Calyx tubular up to 1 cm long with 5 ribs and 5 teeth 2-3 mm long. Flower asymetrically campanulate, the

tube 1-2 cm long, bent approximately half way up in West African, Sudanese and Ethiopian populations but usually well above halfway in East African populations (Parker and Riches, 1993). Corolla lobes 4, one bi-lobed almost erect, the others spreading horizontally, up to 2 cm across, pink with some white markings in the throat. The stigma and stamens are hidden in the tube. The main inflorescence spike may bear up to 100 flowers but only 6-10 are open at any one time. The capsules are up to 1 cm long and each develops several hundred minute seeds, approximately 0.3 mm long by 0.2 mm wide. According to Ma et al. (2004), a single *S. hermonthica* plant can produce as many as 50,000 seeds and remain viable for more than 14 years.

Root System: The root system is weak with little or no ability to absorb materials from the soil, but branches develop from lower nodes of the plant, ramifying and developing secondary haustoria and attachments on contact with other host roots.

According to CABI (2004), *S. hermonthica* is present in Arizona, Florida, Hawaii, and North Carolina. However, NAPIS shows that this plant is exotic to the United States.

Figure 1. Flowers of *S. hermonthica.* Photos courtey of USDA-APHIS, www.bugwood.org

Symptoms/Signs

Striga hermonthica causes characteristic yellowish blotches in the foliage about 1 cm long by 0.5 cm wide. In later stages whole leaves may wilt, become chlorotic and die. Stems are shortened, though leaf number may not be reduced. Inflorescence development is delayed or prevented. Root systems, at least in early stages, may be stimulated, and haustoria 1-2 mm across appear like nodules (CABI, 2004).

Survey

Preferred Method: Surveys for *S. hermonthica* are primary conducted using a visual survey. Infestation of a cereal crop by *S. hermonthica* may be apparent before emergence from the soil, by the chlorotic blotches on the crop foliage.

Uprooting may confirm the presence of the haustoria and young parasite seedlings on the root (CABI, 2004) (Fig. 2). Additionally the flowers are quite characteristic and can be readily observed during an infestation (Fig. 3).

Dugje et al. (2006) used a simple random approach where fields were randomly selected from the four cardinal points (north, east, west, and south) of each community. Five $1m^2$ quadrants were marked out with sticks in a plot along a diagonal transect. Emerged *Striga* plants were identified and counted from each quadrant. Berner et al. (1997) suggest keeping to main roads and use the odometer to stop at precise fixed intervals along the road (*e.g.*, every 20 km). The first fields seen after each stop point should be surveyed.

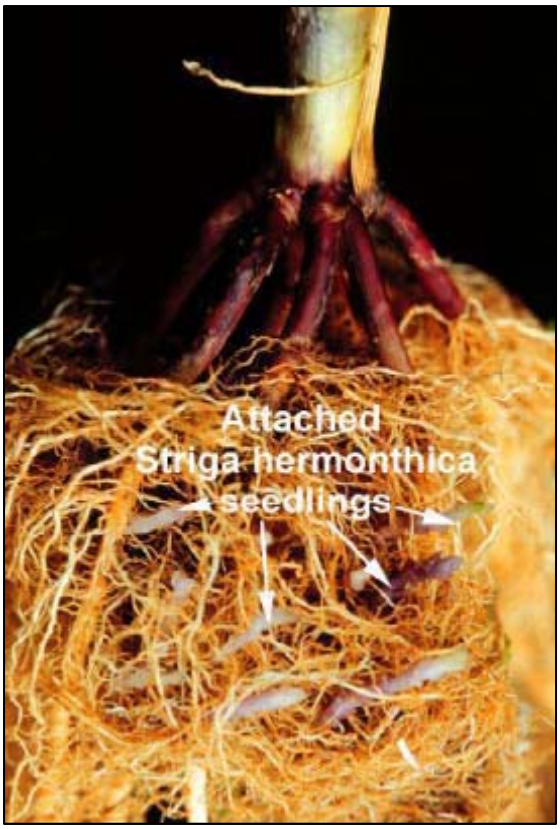

Figure 2. Numerous attached *S. hermonthica* seedlings on a single sorghum root system. Photo from http://www.iita.org/cms/details/striga.pdf

Alternative Method: A revised method for extraction of *Striga* seeds from soil using centrigation and existing techniques based on flotation was described in van Delft et al. (1997). Ten soil cores samples of 3-5 cm diameter and 15 cm depth are suggested within each sample area and mixed together for analysis (Berner et al., 1997).

For detecting the seeds of *S. hermonthica* as a contaminant in crop seeds, Berner et al. (1994) used a technique involving sampling from the bottom of sacks, elutriation of samples in turbulent flowing water and collection of seeds and other particles on a 90 µm

Figure 3. Corn infested with *S. hermonthica.* Photos courtesy of USDA-APHIS.

mesh sieve. *Striga* seeds are then separated from heavier particles by suspension in a solution of potassium carbonate of specific gravity 1.4 in a separating column. Sound seeds collect at the interface and are transferred to a 60 µm mesh for counting (CABI, 2004).

Key Diagnostics

Ramaiah et al. (1983) describe developmental and morphological features to distinguish *Striga* spp. Scanning electron microscopy (SEM) observation of seeds can distriguish some *Striga* spp. and *Buchnera* spp. (Krause and Weber, 1990). However, *S. asiatica*, *S. gesneroides*, *S. hermonthica*, and *S. aspera* are quite difficult to distinguish based on seed coat morphology.

Key to some* Striga *species (Adapted from: Ramaiah et al. 1983)

1a. Calyx ribs 5.....2
1b. Calyx ribs more than 5.....4

2a. Scale-like leaves, short plant stature frequently profusely branched with spiked appearance, flower color variable.....***Striga gesnerioides***
2b. Relatively long slender leaves, coarse textured, with relatively long branches, pink or mauve flowers 8–15 mm across.....3

3a. Flower neck clearly longer than calyx and bent well above calyx. Glandular hairs evident on corolla tube. Leaves narrow.....***Striga aspera***
3b. Flower neck (corolla tube) about equal to calyx in length and bent immediately above calyx. Corolla tube glabrous in appearance…….***Striga hermonthica***

4a. Calyx ribs 10 or more, relatively small and delicate appearing plant, variable flower color…….....***Striga asiatica***
4b. Calyx ribs 1-5, somewhat broad leaves, coarsely toothed, salmon-pink flowers…….....***Striga forbesii***

For additional information on *Striga* spp. see
http://www.iita.org/cms/details/striga.pdf

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Tertiary Pests of Small Grains (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to small grains and exotic to the US

None at this time

Abbreviated Table of Survey Techniques for Small Grains Pests

Insects and Mollusks

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Autographa gamma – silver Y moth	Moth	Leaves skeletonized (young larvae), rolled or folded. Whole leaves eaten or holes in the interior (older larvae). Petioles (leaf stalks) cut. Webbing, frass visible. Damage to leaves, flowers, pods/seeds. Older leaves are preferred. Only eats young leaves after destroying the old ones.	Alternative Method. On individual plants look for: eggs (singly or in small clusters) on both sides of leaves of low-growing plants; Larvae active at night. During the day, larvae remain pressed against the underside of the leaf; when disturbed tend to drop off plant; Pupae found in the folds of the lower leaves of the host plant; Adult moths feed on flowers and can often be seen feeding during the day or early evening. Adult moths have a Y-mark on the forewing that is distinct and silvery.	Preferred Method. Pheromone trap: (Z)-7-dodecenyl acetate and (Z)-7-dodecenol in ratios from 100:1 to 95:5 have been used to monitor male flight. The pheromone may be dispensed from rubber septa at a loading rate of 1 mg. Lures should be replaced every 30 days. Traps should be placed within or on the edge of fields of the host crops. Traps should be suspended from stakes and placed at the level of crop height and raised as the crop matures. This lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory in the 100:1 ratio. Sticky traps and Robinson traps are not recommended.	Due to the migratory nature of this species, adult *A. gamma* can be observed every month from April to November, usually peaking in late summer. Winter: Overwinter as 3rd or 4th instars or in pupal stage. There is no true diapause.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Cernuella virgata – maritime garden snail	Mollusk	Rasping and defoliation of plants. Damage to stalks and top of plants.	Preferred Method. Look on top of plants and structures (fence posts) for: Presence of relatively small snails up to 15 mm (0.59 inches) in diameter with prominent spiral banding on shell; Presence of ribbon-like excrement (slime trails) on plants and structures; Snails are nocturnal with their activity closely linked to moisture availability. Surveys are best carried out at night using a flashlight, or in the morning or evenings following a rain event.	No trap available.	Annual life cycle. Fall/Winter: Breeding Summer: Aestivate on plant heads and stalks. Usually found on top of plants during summertime but have been found feeding on new growth earlier in season

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Cochlicella spp. – pointed snails	Mollusk	Snails cause feeding damage to crop seedlings and legume-based pastures and foul herbage with their slime.	Preferred Method. While conducting a survey, look for clues that suggest the presence of mollusk pests which may include: snails, juveniles and adults, eggs, empty snail shells, mucus and "slime" trails, and/or feces. On rainy or humid days, inspect containers that are suspended inches above the ground, garbage bins, driveways, low-growing bushes, and ground-lying trash. During dry or hot days, snails are attached, often in clusters of several to many individuals, to plants fences, and other objects. Due to this behavior survey during dry weather may actually be easier, especially if snail density is low <u>Alternative Method:</u> Soil litter can also be sampled for mollusks in general.	No trap available.	Biennial life cycle. Summer: Aestivate on plant heads and stalks.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Copitarsia spp.- owlet moths	Moth	Larvae generally feed externally on leaves, stems, and fruits or host plants but will occasionally bore into thicker non-woody tissues	Alternative method. Surveys for *Copitarsia* spp. are generally conducted visually at the ports by examining plant for the presence of egg masses and/or larvae.	Preferred method. A pheromone consisting of (Z)-9-tetradecenyl acetate (Z9-14:Ac) and Z-9-tetradecenol (Z9-14:0H) has been previously identified for *C. decolora*. Captures in traps baited with a mixture of Z9-14:Ac and Z9-14:0H at 4:1, 10:1, and 100:1 ratios were not significantly different from traps baited with virgin females. The commercial availability of this pheromone, however, is unknown at this time. Early detection surveys have traditionally utilized non-selective black light trapping.	May be found throughout the year during the growing season. Winter: Overwinter as pupae in the soil. Diapause has not been reported for the genus.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Helicoverpa armigera- old world bollworm	Moth	In sorghum and other grains, the larvae feed on the head when the grains are in the milky stage. Yield loss is caused by *H. armigera* feeding directly on the grain. In other crop species, bore holes are visible at the base of flower buds, the latter being hollowed out. Leaves and shoots may also be consumed by larvae. Larger larvae bore into maturing flowers, fruit, and seed. Fruit drop and defoliation are possible. Secondary infections by other organisms are common and lead to rotting.	Alternative Method. Look for eggs on/near floral structures (preferred), leaves, and stems; typically found on pubescent (hairy) structures rather than smooth leaf surfaces. Larvae feed on flowers and seed as they develop. Look for terminal leaf damage caused by young larvae, entry holes on floral structures or seed, inside fruit or fruit structures. It may be necessary to cut open the plant organs to detect the pest.	Preferred Method. Traps using (Z)–11-hexadecenal and (Z)-9-hexadecenal in a 97:3 ratio have been used to monitor populations of *H. armigera*. This lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory. This lure utilizes the components in a 25:1 ratio with 10% BHT as a stabilizer is dispensed from a rubber septum and should be replaced every four weeks.	Adults appear in April to May and can be observed until October, because of the migratory nature of this species.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Heteronychus arator – black maize beetle	Beetle	Stems experience external feeding and the whole plant may be toppled or uprooted. Associated organisms: Look for foraging magpies and crows on turf, if present in the understory.	Preferred method. Inspect immediately below the soil surface for signs of attack: frayed chewing around the stem circumference. Seedlings and cuttings may also perish as a result of feeding. In grass and turf, heavy infestations can be detected by lifting up tufts of grass and inspecting for abundant frass or distinct channeling of soil with embedded larvae. Less dense infestations will be evident if sections of grass are dug and examined for presence of larvae or adults. Survey methods developed for potatoes that could be adapted, involve inspecting soil cross sections for the presence of beetles and associated plant damage. Soil sampling has also been used for belowground stages.	Alternative method. Adults fly to light. Light trap captures tend to be female with the greatest number of captures in fall. Males tend to dominate pitfall traps in spring, but captures are reported to be a poor indicator of beetle density.	Univoltine, majority of the lifetime spent underground save some adult flying. Spring to early summer: Majority of mating and most oviposition occurs. Adults crawl on soil surface at night with limited flying. The most damage is observed during this time. Summer/Fall: 3 larval instars, which mature in midsummer. Adults emerge in summer to late autumn with some random adult flying. Winter: Spent as actively feeding, non-reproductive adults. Reproductive maturation occurs slowly.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Lobesia botrana – European grape vine moth	Moth	No specific symptoms are given for small grains. In grape: On inflorescences (first generation), neonate larvae firstly penetrate single flower buds. Symptoms are not evident initially, because larvae remain protected by the top bud. Later, when larval size increases, each larva agglomerates several flower buds with silk threads forming glomerules visible to the naked eye, and the larvae continue feeding while protected inside. Larvae usually make one to three glomerules during their development. Despite hygienic behavior of larvae, frass may remain adhering to the glomerules.	Alternative method. No specific methods are given for small grains. In grape: Coduct a visual inspections for eggs on flower buds or pedicels of vines and grapes. It is preferable to look for larval damage rather than for eggs, because detection of eggs is very tedious and time-consuming, especially under field conditions. Look for webbed bud clusters (glomerules) or flowers where the spring generation larvae feed. Inspect for pupae under rolled leaves in spring. Inspect grapes and look for eggs or damaged berries. Cut open grapes and search for summer generation larvae and pupae.	Preferred method. A pheromone lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory. The lures is loaded wit 0.5 mg of (*E*,*Z*)-(7,9)-dodecadienyl acetate. Traps placed 4 ft high (1.3 m) are generally more effective than traps placed at only 1 ft. Lures for *L. botrana* can be used in the same trap with lures for *Lymantria dispar*, or *Cydia pomonella.* Not recommended. Light traps have been used but their lack of specificity makes their use inadvisable when pheromones are available. A corrugated paper band technique has been used to trap and quantify overwintering pupae.	Adults emerge and begin egglaying in early spring. Usually observed around flowering time.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Nysius huttoni – wheat bug	True bug	The wheat bug is a seed feeder, but may also feed on the foliage. It piercing the grain and sucks out the juices. Damage usually occurs at field edges. When the bug-damaged grain matures, it is characterized by a dark insect-feeding puncture mark surrounded by a pale area on the surface. Some grains shrink to a cuboid shape when much endosperm is removed, presumably from prolonged feeding. Seed germination is not affected by *N. huttoni.* Because the wheat bug injects an enzyme during feeding, flour from damaged wheat ruins dough during breadmaking.	<u>Preferred Method:</u> Bug damage to mature wheat kernels can be easily recognized as pale, slightly elevated patches, often with one or more black or red dots considered to be the marks of bug stylet punctures, or pitting, blackening, and distortion (especially under the microscope). <u>Alternative Method:</u> Soil to a depth of 10 mm plus all plant material was removed from each quadrant (8 randomly placed 0.25m^2 quadrants), bagged, and taken to the laboratory. After agitation in a 25% saline solution and removing the floatant, the retained material was examined under a microscope.	Pheromone traps are not available. A few "flight" traps have been used (see text in datasheet).	Wheat is most vulnerable to damage at the flowering and grain filling stages of growth. The bug attacks wheat kernels in the water ripe to milky ripe stages of development.

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Spodoptera littoralis- Egyptian cotton leafworm	Moth	Damage includes: skeletonized leaves, leaf scars, bare sections on leaves, stripping of plants stems, and buds, and bore holes in stems. Damage to stem may cause plant to wilt distal to entry hole. Damage to stems, leaves, flowers, fruit. Frass present often protruding from bore holes.	Alternative Method. Look for: eggs covered with hair scales on leaves; Early instars (<3rd) on lower leaf surfaces during the day. Larval feeding on leaves, stems, fruit, or pods in any growth stage. Sweep net sampling. 1st-3rd instars detected by sweep net sampling at dawn or dusk; Soil sampling. Later instars (4th – 6th) and pupae may be found by sieving soil samples; Pupae in soil at base of plant.	Preferred Method. Pheromone trap: The synthetic sex pheromone (Z,E) – (9, 11) – tetradecadienyl acetate highly effective for trapping males. Delta traps are placed 1.7 m above the ground at a rate of 2 traps/ha. Pheromone lures impregnated w/ 2 mg of pheromone blend are replaced after 4 weeks of use. A lure is available from the OTIS Pest Survey, Detection, and Exclusion Laboratory. The lure (a 200:1 mixture of (Z, E)-(9-11)-tetradecadieyl acetate to (Z,E)-9,12)-tetradecadienyl acetate is formulated in a Beem capsule with a 2-week field life. For large orders, laminates are formulated with a 12-week field life. Not recommended. Light traps have been used to non-discriminately capture multiple *Spodoptera* spp.	Spring/Summer/Fall: Damage may occur from spring to fall, anytime plants are actively growing

Pest	Type	Symptoms	Visual Survey	Traps	Time of year
Spodoptera litura – rice cutworm	Moth	General Symptoms include: skeletonization of leaves, leaf scars, holes and bare sections are later found on leaves, young stalks, bolls, and buds, stem mining, and wilting. Larvae are leaf eaters but sometimes act as a cutworm with crop seedlings.	Alternative Method. Look for: 'scratch' marks on the leaf surface (upper and middle portion of plant) left by newly emerged larvae. Check for 1st and 2nd instar larvae during the day on the undersurface of leaves and host plants. Third instar and larvae rest in upper soil layers during the day. Watch for skeletonized foliage and perforated leaves and external feeding damage to fruit. Sweep net for adults and larvae at dawn or dusk.	Preferred Method. Pheromone trap: The synthetic sex pheromone (Z,E)-(9,11)-tetradecadienyl acetate and (Z,E)-(9,12)-tetradecadienyl acetate is effective in trapping males. Traps are placed 1-2 m above the ground. The two components in a ratio of 9:1 are available commercially as Litlure in Japan. A lure is available from the CPHST- Otis lab. Not recommended. Light traps have been used to non-discriminately capture multiple *Spodoptera* spp.	Spring/Summer/Fall: Damage may occur from spring to fall, anytime plants are actively growing. Three peak periods of each laying have been observed in the third weeks of June and July and in mid-August.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Heterodera filipjevi	Nematode	Symptoms are often due to mixed population of cereal cyst nematode or other soilborne pathogens and can mimic other problems such as nutrient deficiencies or drought stress. In general, wheat plants become chlorotic and stunted due to the presence of cereal cyst nematodes. Growth may be patchy Barley roots exhibit no readily discernable symptoms. Leaf tips often become discolored: reddish yellow on wheat, red on oats, and yellow on barley. Few or no tillers, rotting of lower culms and crown, and light brown roots with little or no branching were observed in Oregon wheat plants.	Preferred Method: As for other cyst-forming nematodes, in the absence of a host crop, soil samples must be collected and processed to extract cysts. Cysts can be extracted from the subsample using a Fenwick can method, a modified Baermann funnel technique, Cobb's decanting and sieving, sieving decanting and centrifugation, a fluidizing column, a Kort elutriator, and modified Fenwick can elutriation method has also been used with further separation of cysts from the plant debris by flotation in an ethanol and glycerin solution Motile nematodes (*e.g.* juveniles) can be extracted using the Whitehead tray method.	No known vector or associated insects.	In Oregon, patches of stunted seedlings (3-5 leaf stage) appeared in March.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Heterodera latipons - Mediterranean cereal cyst nematode	Nematode	Slight to severe yellowing of cereal stands can be observed at an early stage of nematode infestation. Later, infested fields show patchy plant growth associated with poor tillering and shorter spikes. Symptoms occur in patches that enlarge as the nematode population increases and are similar to those caused by other biotic or and abiotic stresses. Plants also tend to wilt during warmer portions of the days.	Preferred Method: As for other cyst-forming nematodes, in the absence of a host crop, soil samples must be collected and processed to extract cysts. <u>In the presence of a crop:</u> Before plant flowering, white lemon-shaped females can be observed on the roots by the naked eye or under a dissecting microscope after gently shaking or washing the roots to remove adhering soil. However, at a later stage of development, females become easily detached from the roots making the detection of the nematode very difficult even in cases of heavy infestation, especially in non-sandy soils.	Damage is more severe in fields infested concomitantly by *H. latipons* and the fungus *Bipolaris sorokiniana*, the causal agent of common root rot and seedling blight of barley, as the nematode increases the aggressiveness of the fungus.	In the Mediterranean, females are well developed in March; cysts are well developed by April.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Meloidogyne artiellia - British root-knot nematode	Nematode	Plants show impaired root growth, small gall formation, proliferation of lateral roots, and impaired root function. Symptoms, which may be similar to nutrient or water deficiency, include: chlorosis, stunted growth, and/or necrosis of above-ground plant parts. Infested plants may appear wilted under hot and sunny conditions, even with ample soil moisture. Injured root tissue is susceptible to other disease-causing pathogens. In wheat, spikes are sparse and reduced in size. Root galls induced by *M. artiellia* are very small and often are covered by large egg masses that represent the only visible signs of the nematode infection.	Preferred Method. Soil and roots from plants exhibiting symptoms should be collected and the roots examined with the aid of a stereomicroscope for the presence of galls and nematode egg masses adhering to the small galls. Root-knot nematodes are then extracted from soil using a variety of techniques.	In two genotypes of chickpea with complete resistance to Fusarium wilt, infection by *M. artiellia* overcame the resistance to *Fusarium oxysporum* f. sp. *ciceris* race 5.	In climates with cool, wet winters and warm, dry summers, *M. artiellia* is active during spring and winter months and inactive from late spring through summer.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Peronosclerospora philippinensis	Fungus-like	Symptoms of the disease and signs of the pathogen are not listed specifically for oat. On corn, the first symptoms typically appear as chlorotic stripes at the first leaves as early as 9 days after planting. All leaves on a plant may show characteristic symptoms of long chlorotic (yellow) streaks. A downy (grayish) covering primarily on the underside of the leaves is characteristic beginning at the two-leaf stage and is present until the appearance of tassels and silks. As the plant ages, leaves may narrow, become abnormally erect, and appear somewhat dried-out. As the corn plant matures, tassels become malformed and produce less pollen, ear formation is interrupted, and sterility of seeds can result. There are no external symptoms on seeds. If infection occurs early, plants are stunted and may die.	Survey is conducted via visual survey of plants for symptoms of the disease (chlorotic stripes or streaks) and signs of the pathogen (downy growth on underside of leaves consisting of conidia and conidiophores). Spore trapping using Burkhard spore traps and sentinel plots (unsprayed, susceptible plants that are scouted regularly) are suggested for early detection and have been employed for resistance screening.	There is a synergistic relationship between downy mildew fungi and maize streak virus (MSV) on corn. Infection by MSV can mask symptoms of downy mildew infection. Reduction in height and biomass were significantly greater with pathogen combinations than with single pathogens.	Grain fields or forages can be inspected for the disease at any stage of growth. In corn, symptoms are most noticeable and first appear on young leaves.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Puccinia graminis f. sp. *tritici* – Race Ug99 - stem rust of cereals	Fungus	The uredinia of the fungus may occur on leaves, stems, leaf sheaths, spikes, glumes, awns and occasionally on grains of their grassy hosts. Stems and leaf sheaths are the main tissues affected, however. On stems, the uredinia are elongated and reddish-brown. Loose epidermal tissue is conspicuous at the margins of the uredinia, giving a roughened feel to the stem surface. The telial stage occurs in the same tissue as the uredinial stage, but becomes shiny-black.	Preferred Method. Visual survey: The red-brown pustules are easily recognized against the normally blue-green color of healthy stem tissue. However, there may be other causes for similar discoloration. Infections due to stem rust can usually be differentiated from these other causes by gently feeling the plant tissue between the thumb and forefinger for the roughened surface caused by rust infection.	No known vector or associated insects.	Grain fields or forages can be inspected for the disease at any stage of growth. From the seedling stage until about the five- or six-leaf stage, stem rust infections are most obvious on the leaves. The main time for inspection is from the stage when the crop begins to head (late boot stage) until near maturity. Stem rust is generally detected in later April in the southern US, much later than either leaf rust or strip rust. In July, stem rust is generally detectable on trap plots of susceptible winter and spring wheat, barley, and wild grasses in the northern Great Plains.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Rice Hoja Blanca Virus - RHBV	Virus	RHBV-infected rice plants show stunting, chlorotic or yellow stipling (stripes) and mottling of leaves, and premature wilting. In some cases, the whole leaf blade may turn white. Sterility and death of the plant normally follow. The palea and lemma are distorted and turn brown. Infected plants also have fewer and smaller roots, and the roots may become necrotic. Wheat, rye, barley, oats, and some grasses are also hosts for this virus. Wheat becomes infected especially when grown adjacent to rice and shows a characteristic and prominent gray-white discoloration of the spike and uppermost leaves, sometimes referred to as "white tip" or "white spike".	Preferred Method. Wheat and rice plants infected by RHBV can be recognized by using a visual survey for the "white leaf" or leaf mottling symptom. Serology (ELISA) is then used to confirm the diagnosis.	RHBV is transmitted in a persistent manner by the planthopper *Tagosodes orizicolus.*	May be found throughout the year during the growing season

Weeds and Parasitic Plants

Pest	Type	Symptoms	Survey Methods	Associated Crop	Time of year
Arctotheca calendula - capeweed	Weed	Capeweed competes with crop species and can have a detrimental effect on crop growth and yield.	Preferred Method. Visual Survey. Specific survey methods for capeweed are acking. However, weed maps are typically produced from data sampled at discrete intervals on a regular grid. Distinguished by deeply lobed basal leaves, white downy underneath. Area where seeds attach to the head (receptacle) pitted. Petal-like ray florets yellow above, gray-green below with strip-like parts mostly 1.5-2.5 cm long. Disc florets dark purple; seeds covered in pale brown wool and topped by 6-8 short scales.	Wheat, barley, and oats	The optimum time for a survey is when capeweed is at peak flowering.

Pest	Type	Symptoms	Survey Methods	Associated Crop	Time of year
Moraea spp. – cape tulips	Weed	Cape tulips compete with crop and native species and can have a detrimental effect on growth and yield.	Preferred Method. Visual Survey. Specific survey methods for cape tulip are lacking. However, weed maps are typically produced from data sampled at discrete intervals on a regular grid. *M. flacida:* Distinguished by fibrous-sheathed corm at the base of the plant, orange to salmon pink flowers that are yellow in the centre; single leaves and presence of seed in capsules. *M. miniata:* Distinguished by scaly covering around corm at the base of the plant, pink–salmon coloured flowers with a green dotted yellow centre; leaves 2 or 3; seed not produced but plants produce clusters of bulbils in the swollen leaf axils and often small corms (cormils) around the corm.	Oats	The optimum time for a survey is when *Moraea* spp. is at peak flowering.

Pest	Type	Symptoms	Survey Methods	Associated Crop	Time of year
Striga hermonthica – purple witchweed	Parasitic Plant	*S. hermonthica* causes characteristic yellowish blotches in the foliage about 1 cm long by 0.5 cm wide. In later stages whole leaves may wilt, become chlorotic and die. Stems are shortened, though leaf number may not be reduced. Inflorescence development is delayed or prevented. Root systems, at least in early stages, may be stimulated, and haustoria 1-2 mm across appear like nodules.	Preferred Method. Visual Survey. Look for chlorotic blotches on the crop foliage. Uprooting plants and examining roots for haustoria. Pink/purple flowers are characteristic. Alternative Method: Soil sampling for seeds using flotation and centrifugation.	Barley	May be found throughout the year during the growing season

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Appendix B: Glossary of Terms

Abaxial: Concerning the surface of a structure that is turned away from the structure's primary axis, pertaining to the lower surface of a leaf.

Abiotic: Describes nonliving substances or environmental factors.

Achene: A small dry one-seeded fruit in which the ovary wall is free from the seed

Agglomerates: To form or collect into a rounded mass.

Adaxial: Located on the side or directed toward the axis, pertaining to the upper surface of a leaf.

Aeciospores: Dikaryotic spore of a rust fungus produced in an aecium; in heteroecious rusts, a spore stage that infects the alternate host.

Aecium (pl. aecia; adj. aecial): the fruiting body of a rust fungus in which the first dikaryotic spores (aeciospores) are produced.

Aestivation: Dormancy in summer or during periods of continued high temperatures, or during a dry season.

Allogamous: Relating to cross-fertilization in plants.

Alternate host: A plant other than the main host that a parasite can colonize; alternative hosts are not required for completion of the developmental cycle of the parasite.

Ambisense: Applied to single-stranded RNA viral genomes; part of the nucleotide sequence is of positive-sense, part is of negative-sense.

Androconia: Modified wing scales on butterflies and moths that release pheronomes (also called scent scales).

Annual: A plant that completes its life cycle and dies within one year (see biennial, perennial).

Basidiospores: Haploid (1N) sexual spore produced on a basidium.

Basidium (pl. basidia; adj. basidial): Specialized cell or organ, often club-shaped, in which karyogamy and meiosis occur, followed by production of externally-borne basidiospores (generally four) that are haploid. There are several types of basidia.

Biennial: A plant that produces seed and dies at the end of its second year of growth (see annual, perennial).

Biomass: The total weight of living organisms.

Biotic: Relating to life, as disease caused by living organisms

Brachypterous: Having short wings that do not cover the abdomen.

Calcareous: Containing calcium.

Chlorosis (adj. chlorotic): Failure of chlorophyll development, caused by disease or a nutritional disturbance; fading of green plant color to light green, yellow, or white

Chorion: The outer shell or covering of the insect egg.

Clutch: The number of eggs layed in a given time period.

Coalesce: Grow together into one body or spot

Conidium (pl. conidia): An asexual, nonmotile fungal spore that develops externally or is liberated from the cell that formed it.

Conidiophores: Simple or branched hypha on which conidia are produced.

Contaminate: To make impure or unclean by contact.

Corm: The rounded underground storage organ and stem of plants such as crocus that resemble a bulb.

Cryptic: Serving to conceal, hide.

Cypsela: A one-seeded, one-celled, indehiscent fruit; an achene with the calyx tube adherent

Cyst: In fungi, a resting structure in a protective membrane or shell-like enclosure. In nematodes, the egg-laden carcass of a female nematode.
In bacteria, a specialized type of bacterial cell enclosed in a thick wall, often dormant and resistant to environmental conditions.

Defoliation: Loss of leaves from a plant, whether normal or premature.

Degree Days: Development of poikilothermic ("cold-blooded") organisms such as insects, fungi, and plants, is regulated by environmental temperatures.

Development to particular stages in the life cycles of these organisms is largely controlled by how much heat they experience, where heat is considered as a function of temperature and time. Degree-days are an estimate of the amount of heat accumulated over a 24-hr period. They are calculated using lower and upper developmental thresholds unique to a particular organism and, typically, some approximation of the 24-hour temperature pattern derived from minimum and maximum daily temperatures (which are commonly available from local recording weather stations). Only those temperatures falling between the lower and upper thresholds are included in the calculations. Degree-day values may be positive or equal zero (all temperatures above or below thresholds), but are never negative. Degree-days are calculated for each day and are then summed to provided cumulative (total) degree-days. Starting points for calculating cumulative degree-days are usually arbitrary, typically January 1 but often later (e.g. April 1) in areas with cold winter temperatures. Based on experimental data, cumulative degree-days are linked to specific development events of interest (e.g. adult insect emergence). Thus, a pest manager can anticipate or predict an event of interest based on local temperature data and an appropriate degree-day based developmental model (Hansen, 2007).

Dehiscent: 1) Botany: Splitting open at maturity to release contents (of a fruit); 2) Pathology: of an ascus or fruit-body, opening when mature, by a pore or by rupturing or fragmentation; of conidia and other spores, falling off.

Demicyclic: A rust fungus that lacks the urediniospore (repeating) stage (e.g. many species of *Gymnosporangium*) (see macrocyclic, microcyclic).

Diapause: A condition of restrained development and reduced metabolic activity, which cannot be directly attributed to unfavorable environmental conditions. Regarded by entomologists to involve a resting period of an insect, especially of larvae in winter (hibernation, quiescence).

Dichotomous: Dividing into two equal branches

Diploid: Having two complete sets of chromosomes (2N chromosomes) (see haploid).

Disease: Abnormal functioning of an organism

DNA (abbr. for deoxyribonucleic acid): The double-stranded, helical molecule that contains genetic code information. Each repeating unit, or nucleotide, is composed of deoxyribose (a sugar), a phosphate group, and a purine (adenine or guanine) or a pyrimidine (thymine or cytosine) base

Dorsal: On the upper surface.

Ectoparasite: Parasite that feeds from the exterior of its host (see endoparasite).

Elutriation: The operation of pulverizing substances and mixing them with water in order to separate the heavier constituents, which settle out in solution, from the lighter constituents.

Endoparasite: parasitic organism that lives and feeds from inside its host (see ectoparasite).

ELISA (Enzyme-Linked ImmunoSorbent Assay): A serological test in which the sensitivity of the reaction is increased by attaching an enzyme that produces a colored product to one of the reactants.

Facultative diapause: May or may not need to diapause; not required for development.

Fallow: Cultivated land kept free from a crop or weeds during the normal growing season.

Fecundity: The number of offspring per number of potential offspring (e.g. eggs).

Fenestration: For nematodes, a clear area around the anus of the cyst.

Filament: A slender threadlike structure.

Fitness: The ability of an organism to survive and reproduce; the ability of an organism to pass its genes to the next generation.

Floret: One of the small, closely clustered flowers forming the head of a composite flower.

Forage: Eatage: animal food for browsing or grazing.

Frass: Plant fragments made by a wood-boring insect usually mixed with excrement; solid larval insect excrement.

Gall: An abnormal swelling or localized outgrowth, often roughly spherical, produced by a plant as a result of attack by a fungus, bacterium, nematode, insect, or other organism

Gelatinous: Rubbery, jelly-like.

Giant Cell: Enlarged, multinucleate cell formed in roots by repeated nuclear division without cell division induced by secretions of certain sedentary plant-parasitic nematodes.

Glomerule: A very dense cluster.

Haploid: Having a single complete set of chromosomes (see diploid).

Haustorium (pl. haustoria): Specialized branch of a parasite formed inside host cells to absorb nutrients.

Herbaceous: Describing primary, soft, nonwoody tissue, as a plant or plant part; having the characteristics of an herb.

Herbage: Succulent herbaceous vegetation of pasture land.

Hermaphrodite (adj. hermaphroditic): Having both male and female reproductive organs.

Hyaline: Transparent or nearly so; translucent; often used in the sense of colorless.

Incubation period: The time between penetration of a host by a pathogen and the first appearance of disease symptoms.

Inflorescence: A characteristic arrangement of flowers on a stem. A flower cluster.

Inoculum: Pathogen or its parts, capable of causing infection when transferred to a favorable location.

Instar: Refers to the larva during the period between molts, numbered to designate the various periods (e.g., the first instar is the stage between the egg and the first molt).

Iridescent: A display of lustrous rainbow-like colors.

Isozymes: The different forms of an enzyme that carry out the same enzymatic reaction but require different conditions (pH, temperature, etc.) for optimum activity.

Lemma: The lower of two bracts enclosing the flower in grasses.

Lesion: Localized diseased area or wound.

Looper: A caterpillar that moves by looping (placing the rear end of the body next to the thorax before extending the front part) its body.

Lure: An attractant.

Macrocyclic: A rust fungus that typically exhibits all five stages of the rust life cycle (see demicyclic, microcyclic).

Macropterous: With well developed long wings.

Mating Type: Compatible strains, usually designated + and - or A and B, necessary for sexual reproduction in heterothallic fungi.

Meiosis: Process of nuclear division in which the number of chromosomes per nucleus is halved (i.e. converting the diploid state to the haploid state) (see mitosis).

Microcyclic: Describing a rust fungus that produces only teliospores and basidiospores (see demicyclic, macrocyclic).

Micropile: 1) Botany: a very small opening in the outerr coat of an ovule, through which the pollen tube penetrates; the corresponding opening in the developed seed; 2) Entomology: one of the minute openings in the insect egg, through which spermatozoa enter in fertilization.

Microplot: Microplots are enclosures that allow plant growth in limited volumes of field soil which is physically isolated from ambient soil. By definition, microplot systems are implemented under field or near-field conditions. Dimensions should be large enough to allow rootgrowth similar (not identical) to that which occurs under natural conditions. Microplots facilitate study of interactions between soil-borne pathogens and their plant hosts under biotic and environmental conditions which are more realistic than greenhouse conditions

Migratory: An insect, nematode that moves from place to place on a plant, from plant to plant when feeding, or across regions (see sedentary).

Mitosis: Nuclear division in which the chromosome number remains the same (see meiosis).

Morph: One of various distinct forms of an organism or species.

Mosaic: Disease symptom characterized by non-uniform coloration, with intermingled normal, light green and yellowish patches, usually caused by a virus; often used interchangeably with mottle.

Mottle: Disease symptom comprising light and dark areas in an irregular pattern, usually caused by a virus; often used interchangeably with mosaic.

Multivoltine: Pertaining to organisms with many generations in a year or season.

Necrotic: Death of cells or tissue, usually accompanied by black or brown darkening.

Nematode: Nonsegmented roundworm (animal), parasitic on plants or animals, or free living in soil or water.

Neonate: Newly born individual.

Nocturnal: Active during the night.

Obligate: Restricted to a particular set of environmental conditions, without which an organism cannot survive (e.g., an obligate parasite can survive only by parasitizing another organism.)

Ocellus (pl. ocelli): The simple eyes or stemmata of insects, usually situated on the crown of the head between the great compound eyes.

Oospore: Thick-walled, sexually-derived resting spore of oomycetes.

Overwinter: To survive or persist through the winter period.

Oviposit (oviposition): To deposit or lay eggs or ova. The act of depositing eggs.

Palea: In grasses, the uppermost bract enclosing the flower or caryopsis.

Panicle: An inflorescence with a main stem and branches, the flowers on the lower branches open earlier than the upper ones.

Parasite (adj. parasitic): Organism that lives in intimate association with another organism on which it depends for its nutrition; not necessarily a pathogen

PCR (acronym for polymerase chain reaction): A technique used to amplify the number of copies of a specific region of DNA in order to produce enough of the DNA for use in various applications such as identification and cloning.

Perennial: Something that occurs year after year; plant that survives for several to many years (see annual, biennial).

Perineal Pattern: Fingerprint-like pattern formed by cuticular striae surrounding the vulva and anus of the mature nematode females.

Persistent transmission (syn. circulative transmission): A type of virus transmission in which the virus is acquired and transmitted by the vector after relatively long feeding times and remains transmissible for a prolonged period while in association with its vector

Petiole: Stalk portion of a leaf.

Pheromone: A substance given off by one individual that causes a specific reaction by other individuals of the same species, such as sex attractants, alarm substances, etc.

Photoperiod: The length of time that light is present in a given time interval, such as a daily 24-hour time interval.

Phytophagous: Plant eating.

Polymorphic: Literally meaning having more than one form.

Polyphagous (Polyphagy): Eating many kinds of food.

Predispose: To make prone to infection and disease.

Prolegs: Fleshy unjointed abdominal legs of insect larvae.

Propagative transmission (syn. circulative propagative transmission): Pathogen transmission characterized by a long period of acquisition of the pathogen (usually a mollicute, e.g. phytoplasma or spiroplasma, and sometimes a virus) by a vector (typically an insect), a latent period before the vector is able to transmit the pathogen, and retention of the pathogen by the vector for a long period because the pathogen reproduces or replicates in the vector.

Quiescent: Not active, quiet.

Race: Subgroup or biotype within a species or variety, distinguished from other races by virulence, symptom expression, or host range, but not by morphology.

Random Amplified Polymorphic DNA (RAPD): A technique using single, short (usually 10-mer) synthetic oligonucleotide primers for PCR. The primer, whose sequence has been chosen at random, initiates replication at its complementary sites on the DNA, producing fragments up to about 2 kb long, which can be separated by electrophoresis and stained with ethidium bromide. A primer can exhibit polymorphism between individuals, and polymorphic fragments can be used as markers.

Rasping: Chewing.

Real Time PCR: A laboratory technique based on polymerase chain reaction, which is used to amplify and simultaneously quantify a targeted DNA molecule. It enables both detection and quantification (as absolute number of copies or relative amount when normalized to DNA input or additional normalizing genes) of a specific sequence in a DNA sample. The procedure follows the general principle of polymerase chain reaction; its key feature is that the amplified DNA is

quantified as it accumulates in the reaction in *real time* after each amplification cycle.

Receptive hypha: The part of a rust fungus pycnium (spermogonium) that receives the nucleus of a pycniospore (spermatium)

Resistant (n. resistance): Possessing properties that prevent or impede disease development (see susceptible)

Restriction fragment length polymorphism (RFLP): A variation in DNA sequence that is easily recognized because it occurs at a site where a restriction enzyme cuts a specific sequence, producing DNA fragments of varying lengths. RFLP's often serve as genetic markers.

Reticulation: Resembling or forming a net or network.

Ribonucleic acid (abbr. RNA): Several nucleic acids composed of repeating units of ribose (a sugar), a phosphate group, and a purine (adenine or guanine) or a pyrimidine (uracil or cytosine) base; transcribed from DNA and involved in translation to proteins.

Sclerotized: Hardened.

Sedentary: Not active; settled or remaining in one place.

Semi-looper: A caterpillar in which 1-2 pairs of the abdominal legs are absent and movement is restricted to progression only in small loops (of the Noctuoidea superfamily.)

Sentinel plot: Unsprayed, susceptible plants that are scouted regularly as part of an early pest detection program.

Sequence Characterized Amplified Region (SCAR): SCARs are DNA fragments amplified by the Polymerase Chain Reaction (PCR) using specific 15-30 bp primers, designed from nucleotide sequences established in cloned RAPD (Random Amplified Polymorphic DNA) fragments linked to a trait of interest. By using longer PCR primers, SCARs do not face the problem of low reproducibility generally encountered with RAPDs. Obtaining a codominant marker may be an additional advantage of converting RAPDs into SCARs.

Serology: A method using the specificity of the antigen-antibody reaction for the detection and identification of antigenic substances and the organisms that carry them.

Sessile: Not supported on a stem or footstalk; immobile.

Sexual dimorphism: Sexes are different in form or color in the same species; may be seasonal or geographic; male and female look different by color form etc.

Sieve: A strainer for separating particles of various sizes.

Sign: Indication of disease from direct observation of a pathogen or its parts (see symptom)

Skeletonize: To remove leaf tissue between the veins, leaving the network of veins intact.

Solitary: Lone.

Spermagonium (pl. spermagonia; syn. pycnium for rust fungi): Structure in which male reproductive cells are produced; in rust fungi, globose or flask-shaped haploid fruiting body composed of receptive hyphae and spermatia (pycniospores).

Spermatium (pl. spermatia; syn. pycniospore for rust fungi): A male sex cell; a nonmotile male gamete; a haploid male gamete.

Spermatophore: A bundle used to transfer sperm.

Spore: A specialized reproductive body in fungi (and some other organisms), containing one or more cells, capable of developing into an adult.

Stippling: Series of small dots or speckles in which chlorophyll is absent.

Stunting: Reduction in height of a vertical axis resulting from a progressive reduction in the length of successive internodes or a decrease in their number.

Susceptible: Prone to develop disease when infected by a pathogen (see resistance).

Symptoms: The external and internal reactions or alterations of a plant as a result of disease.

Syncytium (pl. syncytia): A multinucleate structure in root tissue formed by dissolution of common cell walls induced by secretions of certain sedentary plant-parasitic nematodes (e.g. cyst nematodes).

Synergism: Greater than additive effect of interacting factors.

Synonymy: A section of a systematic presentation about an organism that lists all of the names that have been used for the organism including synonyms, new

combinations, misidentifications, etc. In some cases this section may include only true synonyms.

Tegumen: Lepidoptera: the tergum in male genitalia. A structure shaped as a hood or inverted trough, positioned dorsad of the anus; the uncus articulates with its caudal margin, derived from the ninth abdominal tergum.

Teliospore (sometimes called teleutospore, teleutosporodesm): Thick-walled resting or overwintering spore produced by the rust fungi (Uredinales) and smut fungi (Ustilaginales) in which karyogamy occurs; it germinates to form a promycelium (basidium) in which meiosis occurs.

Telium (pl. telia): Fruiting body (sorus) of a rust fungus that produces teliospores

Transient: One who stays for only a short time.

Translucent: Shining through; transparent, letting light pass but diffusing it so that objects on the other side cannot be clearly distinguished.

Transovarial transmission: The transmission of microorganisms between generations of hosts via the eggs (vertical transmission).

Transverse: Pertaining to structures which are wider than long; running across or cutting the longitudinal axis at right angles.

Trematodes: A fluke; parasitic flatworms having external suckers for attaching to a host.

Trichome: A plant epidermal hair, of which several types exist.

Truck farming: A horticultural practice of growing one or more vegetable crops on a large scale for shipment to distant markets. At first this type of farming depended entirely on local or regional markets. As the use of railroads and large-capacity trucks expanded and refrigerated carriers were introduced, truck farms spread to the cheaper lands of the West and South, shipping seasonal crops to relatively distant markets where their cultivation is limited by climate. The major truck-farming areas are in California, Texas, Florida, along the Atlantic Coastal Plain, and in the Great Lakes area. Centers for specific crops vary with the season. Among the most important truck crops are tomatoes, lettuce, melons, beets, broccoli, celery, radishes, onions, cabbage, and strawberries.

Urediniospore (also urediospore, uredospore): The asexual, dikaryotic, often rusty-colored spore of a rust fungus, produced in a structure called a uredinium; the "repeating stage" of a heteroecious rust fungus(*i.e.*, capable of infecting the host plant on which it is produced).

Uredinium (also uredium; pl. uredinia): Fruiting body (sorus) of rust fungi that produces urediniospores.

Vector: Literally a bearer; specifically a host of a disease transmissible to another species of organism.

Ventral: Pertaining to the under surface of abdomen.

Vermiform: Worm-shaped

Virulent: Highly pathogenic; having the capacity to cause severe disease.

Virus: A submicroscopic, intracellular, obligate parasite consisting of a core of infectious nucleic acid (either RNA or DNA) usually surrounded by a protein coat.

Wilt: Drooping of leaves and stems from lack of water (inadequate water supply or excessive transpiration); vascular disease that interrupts normal water uptake.

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Appendix C: FY08 CAPS Priortized Pest List and Commodity Matrix

FY08 CAPS Prioritized Pest List and Commodity Matrix

Rank	Scientific Name	Common/Disease Name	Corn (*Zea* spp.)	Soybeans (*Glycine* spp.)	Wheat (*Triticum* spp.)	Cotton (*Gossypium* spp.)	Tomatoes (*Lycopersicon* spp.)	Grapes (*Vitis* spp.)	Potatoes (*Solanum* spp.)	Apples (*Malus* spp.)	Citrus (*Citrus* spp.)	Peanuts (*Arachis* spp.)	Lettuce (*Lactuca* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Barley (*Hordeum* spp.)	Strawberries (*Fragaria* spp.)	Almonds (*Prunus dulcis*)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Carrots (*Daucus carota*)	Cucumbers (*Cucumaria* spp.)	Beans (*Phaseolus* spp.)	Sunflower (*Helianthus* spp.)	Pears (*Pyrus* spp.)	Celery (*Apium graveolens*)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Oats (*Avena* spp.)	Asparagus (*Asparagus* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
1	*Phytophthora ramorum*	Sudden Oak Death									■																					■	■	■	3
2	*Helicoverpa armigera*	Old World Bollworm	■	■	■	■	■		■	■	■	■	■	■	■	■	■	■		■	■	■		■	■			■	■	■		■	■	■	■	26
3	*Planococcus minor*	Passionvine Mealybug						■	■		■																									3
4	*Dendrolimus superans sibiricus*	Siberian Silk Moth																														■	■			2
5	*Ceroplastes destructor*	Soft Wax Scale							■		■															■								■	■	5
6	*Ralstonia solanacearum*	Bacterial Wilt of Potato					■		■															■												3
7	*Achatina fulica*	Giant African Snail									■	■	■									■		■				■	■							7
8	*Unaspis yanonensis*	Arrowhead Scale									■																									1
9	*Eudocima fullonia*	Fruit Piercing Moth					■	■	■	■	■										■								■						■	8
10	*Xanthomonas axonopodis* pv. *citri*	Citrus Canker									■																									1
11	*Xylella fastidiosa* CVC strain	Citrus Variegated Chlorosis						■			■																									2
12	*Adoxophyes orana*	Summer Fruit Tortrix Moth		■		■		■	■	■	■	■					■				■				■								■	■	12
13	*Scirtothrips dorsalis*	Chilli Thrips	■	■		■	■	■	■		■	■					■		■	■	■	■		■	■	■			■		■			■	■	19
14	*Ceroplastus japonicus*	Japanese Wax Scale								■	■										■					■								■	■	6
15	*Oxycarenus hyalinipennis*	Cotton Seed Bug	■			■		■																		■								■	■	6
16	*Agrilus biguttatus*	Oak Splendour Beetle																																■	■	2
17	*Platypus quercivorus*	Oak Ambrosia Beetle																																■		1
18	*Meloidogyne fallax*	False Columbia Root-knot Nematode					■		■				■									■														4
19	*Meloidogyne artiellia*	British Root-knot Nematode			■										■	■												■								4
20	*Ditylenchus angustus*	Rice Stem Nematode												■																						1
21	*Candidatus* Liberibacter africanus	Citrus Greening (African Strain)									■																									1
22	*Heterodera latipons*	Mediterranean Cereal Cyst Nematode			■																									■						2
23	*Melastoma malabathricum*	Banks Melastoma																																■	■	2
24	*Candidatus* Liberibacter asiaticus	Citrus Greening (Asian Strain)									■																									1
25	*Phytophthora quercina*	Oak Decline																																■		1
26	*Lymantria mathura*	Pink Gypsy Moth								■																■								■	■	4
27	*Cochlicella* spp.				■												■					■		■								■	■	■	■	8
28	*Onopordum acaulon*	Horse Thistle																																		0
29	*Tomicus destruens*	Pine Shoot Beetle																														■				1
30	*Thaumatotibia leucotreta*	False Codling Moth	■			■					■					■					■			■									■	■	8
31	*Dendrolimus pini*	Pine-tree Lappet																														■	■			2
32	*Leucoptera malifoliella*	Pear Leaf Blister Moth								■																■									■	3
33	*Chilo suppresalis*	Asiatic Rice Borer	■												■	■																				3
34	*Paratachardina lobata lobata*	Lobate Lac Scale									■															■								■	■	5
35	*Monochamus sutor*	Small White-marmorated Longhorned Beetle																														■	■		■	3
36	*Uromyces transversalis*	Gladiolus Rust																																		0
37	*Citrus tristeza virus* (CTV)	Citrus Tristeza Disease									■																									1
38	*Heterodera cajani*	Pigeonpea Cyst Nematode																						■												1
39	*Ophiostoma longicollum*																																		■	1
40	*Monochamus saltuarius*	Japanese Pine Sawyer																														■	■			2

FY08 CAPS Prioritized Pest List and Commodity Matrix

Rank	Scientific Name	Common/Disease Name	Corn (*Zea* spp.)	Soybeans (*Glycine* spp.)	Wheat (*Triticum* spp.)	Cotton (*Gossypium* spp.)	Tomatoes (*Lycopersicon* spp.)	Grapes (*Vitis* spp.)	Potatoes (*Solanum* spp.)	Apples (*Malus* spp.)	Citrus (*Citrus* spp.)	Peanuts (*Arachis* spp.)	Lettuce (*Lactuca* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Barley (*Hordeum* spp.)	Strawberries (*Fragaria* spp.)	Almonds (*Prunus dulcis*)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Carrots (*Daucus carota*)	Cucumbers (*Cucumaria* spp.)	Beans (*Phaseolus* spp.)	Sunflower (*Helianthus* spp.)	Pears (*Pyrus* spp.)	Celery (*Apium graveolens*)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Oats (*Avena* spp.)	Asparagus (*Asparagus* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
41	*Spodoptera littoralis*	Egyptian Cottonworm	■	■	■	■	■	■	■	■	■	■	■	■	■		■		■		■		■	■	■	■	■	■		■		■	■	■	26
42	*Xanthomonas oryzae* pv. *oryzicola*	Bacterial Leaf Streak of Rice												■																					1
43	*Archips xylosteanus*	Variegated Golden Tortrix							■	■	■										■					■						■	■	■	8
44	*Heterodera sacchari*	Sugar Cane Cyst Nematode												■																					1
45	*Cernuella* spp.			■	■											■								■	■						■	■	■	■	9
46	*Meloidogyne paranaensis*	Parana Coffee Root-knot Nematode					■																												1
47	*Xyleborus dispar*	Ambrosia Beetle						■		■											■					■						■	■	■	■	8
48	*Sirex noctilio*	European Woodwasp																														■	■			2
49	*Leptographium truncatum*	Root Disease																														■	■			2
50	*Meloidogyne mingnanica*	Citrus Root-knot Nematode									■																									1
51	*Meloidogyne fujianensis*	Citrus Root-knot Nematode									■																									1
52	*Phytophthora alni*	Alder Root Rot																																■		1
53	*Homeria* spp.	Cape Tulips																												■						1
54	*Meloidogyne indica*	Citrus Root-knot Nematode									■																									1
55	*Meloidogyne citri*	Citrus Root-knot Nematode					■				■																									2
56	*Meloidogyne jianyangensis*	Citrus Root-knot Nematode									■																									1
57	*Meloidogyne kongi*	Citrus Root-knot Nematode									■																									1
58	*Meloidogyne donghaiensis*	Citrus Root-knot Nematode									■																									1
59	*Protopulvinaria longivalvata*																																■			1
60	*Dendroctonus micans*	Great Spruce Bark Beetle																														■	■			2
61	*Pulvinaria polygonata*	Cottony Citrus Scale									■																									1
62	*Theba pisana*	White Garden Snail									■								■																■	3
63	*Epiphyas postvittana*	Light Brown Apple Moth					■	■	■	■	■						■				■			■		■		■				■	■	■	■	14
64	*Ostrinia furnacalis*	Asian Corn Borer	■			■									■																					3
65	*Scolytus intricatus*	European Oak Bark Beetle																																■	■	2
66	*Hylobius abietis*	Large Pine Weevil																														■	■		■	3
67	*Tropilaelaps clareae*	Bee Mite																																		0
68	*Guignardia citricarpa*	Citrus Black Spot	■								■																								■	3
69	*Striga asiatica*	Asiatic Witchweed	■											■	■																					3
70	*Striga gesnerioides*	Cowpea Witchweed												■																						1
71	*Striga hermonthica*	Purple Witchweed	■											■	■	■																				4
72	*Hylurgus ligniperda*	Red Haired Pine Bark Beetle																														■	■			2
73	*Acacia nilotica*	Egyptian Thorn																																		0
74	*Trioza erytreae*	African Citrus Psyllid									■																									1
75	*Pomacea* spp.	Apple Snails												■																						1
76	*Monochamus alternatus*	Japanese Pine Sawyer Beetle								■																						■	■		■	4
77	*Cecidophyopsis ribis*	Black Currant Gall Mite																																		0
78	*Crocidolomia binotalis*	Cabbage Cluster Caterpillar																											■							1
79	*Chilecomadia valdiviana*	Carpenter Worm								■																■								■	■	4
80	*Autographa gamma*	Silver Y Moth	■	■	■	■	■	■	■	■			■			■				■		■		■	■		■	■		■			■	■	19

FY08 CAPS Prioritized Pest List and Commodity Matrix

Rank	Scientific Name	Common/Disease Name	Corn (*Zea* spp.)	Soybeans (*Glycine* spp.)	Wheat (*Triticum* spp.)	Cotton (*Gossypium* spp.)	Tomatoes (*Lycopersicon* spp.)	Grapes (*Vitis* spp.)	Potatoes (*Solanum* spp.)	Apples (*Malus* spp.)	Citrus (*Citrus* spp.)	Peanuts (*Arachis* spp.)	Lettuce (*Lactuca* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Barley (*Hordeum* spp.)	Strawberries (*Fragaria* spp.)	Almonds (*Prunus dulcis*)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Carrots (*Daucus carota*)	Cucumbers (*Cucumaria* spp.)	Beans (*Phaseolus* spp.)	Sunflower (*Helianthus* spp.)	Pears (*Pyrus* spp.)	Celery (*Apium graveolens*)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Oats (*Avena* spp.)	Asparagus (*Asparagus* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
81	*Orobanche aegyptiaca*	Egyptian Broomrape				■	■		■			■									■			■		■		■							8
82	*Orobanche cernua*	Nodding Broomrape											■											■											2
83	*Vitex rotundifolia*	Roundleaf Chastetree																																	0
84	*Phellinus noxius*	Brown Root Rot																														■		■	■	3
85	*Stenchaetothrips biformis*	Rice Thrips	■											■																						2
86	*Orobanche crenata*	Broomrape					■					■	■									■			■		■		■							7
87	*Xyleborus maiche*	Ambrosia Beetle																																■	■	2
88	*Planococcus lilacinus*	Coffee Mealybug						■	■		■																									3
89	*Urocerus gigas gigas*	Horntail																														■	■			2
90	*Onopordum illyricum*	Illyrian Thistle																																		0
91	*Ephelis oryzae*	Rice Udbatta Disease												■	■																					2
92	*Rhabdoscelus obscurus*	Sugar Cane Weevil	■																																	1
93	*Acacia cyclops*	Coastal Wattle								■	■								■		■					■									■	6
94	*Arctotheca calendula*	Namaqualand Daisy			■											■														■						3
95	*Eutetranychus orientalis*	Citrus Brown Mite	■	■		■		■	■	■	■				■		■	■			■			■	■	■			■				■	■	■	18
96	*Euphorbia terracina*	False Caper									■							■																	■	3
97	*Sarasinula plebeia*	Bean Slug					■		■				■												■			■								5
98	*Synanthedon myopaeformis*	Red-belted Clearwing								■																■								■	■	4
99	*Alectra vogelii*	Yellow Witchweed		■								■												■												3
100	*Elsinoe batatas*	Stem and Leaf Scab																																		0
101	*Chlorophorus strobilicola*	Pine Cone Cerambycid																														■				1
102	*Gymnocoronis spilanthoides*	Senegal Tea Plant												■																						1
103	*Aeginetia indica*	Ye Gu												■																						1
104	*Aleurocanthus spiniferus*	Spiny Black Fly						■			■															■								■	■	5
105	*Cuscuta australis*	Australian Dodder									■											■													■	3
106	*Rice hoja blanca virus* (RHBV)	Rice Hoja Blanca Disease			■									■		■														■						4
107	*Xylosandrus mutilatus*	Ambrosia Beetle																																■	■	2
108	*Phoma tracheiphila*	Mal Secco									■																									1
109	*Cuscuta monogyna*	Dodder																																		0
110	*Pericyma cruegeri*	Poinciana Looper																																		0
111	*Cuscuta reflexa*	Dodder									■											■													■	3
112	*Ageratina riparia*	Mistflower																																		0
113	*Actinoscirpus grossus*	Giant Bulrush												■																						1
114	*Rhynchophorus palmarum*	Palm Weevil									■																							■		2
115	*Copitarsia* spp.	Copitarsia Moths	■		■		■	■	■	■			■				■			■		■			■			■			■				■	14
116	*Metamasius* spp.	Metamasius Weevils	■												■																					2
117	*Philephedra broaduayi*																																			0
118	*Coconut cadang-cadang viroid* (CCCVd)	Coconut Cadang-Cadang Disease																																		0
119	*Acroceras zizanioides*	Oat Grass												■																		■	■	■	■	5
120	*Rubus alceifolius*	Giant Bramble																														■	■	■	■	4

Appendix D: FY09 CAPS Priortized Pest List and Commodity Matrix

AHP Prioritized Pest List for FY09 - Prioritized

Rank	Scientific Name	Common Name	Almonds (*Prunus dulcis*)	Apples (*Malus* spp.)	Asparagus (*Asparagus* spp.)	Barley (*Hordeum* spp.)	Beans (*Phaseolus* spp.)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Carrots (*Daucus carota*)	Celery (*Apium graveolens*)	Citrus (*Citrus* spp.)	Corn (*Zea* spp.)	Cotton (*Gossypium* spp.)	Cucumbers (*Cucumis* spp.)	Grapes (*Vitis* spp.)	Lettuce (*Lactuca* spp.)	Oats (*Avena* spp.)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Peanuts (*Arachis* spp.)	Pears (*Pyrus* spp.)	Potatoes (*Solanum* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Soybeans (*Glycine* spp.)	Strawberries (*Fragaria* spp.)	Sunflower (*Helianthus* spp.)	Tomatoes (*Lycopersicon* spp.)	Wheat (*Triticum* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
1	*Helicoverpa armigera*	Old World bollworm		■		▲	▲	■	■	■		▲	▲	▲	■		■	▲	▲	■	▲		▲	■	▲	▲	■	▲	■	▲	▲	■	■	■	27
2	*Planococcus minor*	Passionvine mealybug			■		■	■	■		■	▲	■	■	■	▲						■	■	▲	■		■	■	■	■			■	■	■	21
3	*Nysius huttoni*	New Zealand wheat bug				■		▲									■	■					■					■	■		▲					8
4	*Dendrolimus superans sibiricus*	Siberian silk moth																														▲	▲			2
5	*Ceroplastes destructor*	Soft wax scale		■								▲											■	■										■	■	6
6	*Spodoptera litura*	Cotton cutworm		▲	■		▲	▲	■	■	■	▲	▲	▲	■	▲	■			▲	■	▲	■	▲	▲	▲	▲	▲	▲	▲	■		■	■	▲	28
7	*Ralstonia solanacearum* race 3 biovar 2	Bacterial wilt				■							■								■		▲							▲						5
8	*Achatina fulica*	Giant African snail		■		■	■	▲	■			■	■	■	■	■	■			■		▲		■	■	■	■		■	■				■	■	21
9	*Unaspis yanonensis*	Arrowhead scale										▲																								1
10	*Eudocima fullonia*	Fruit piercing moth		■					■			■			■	■					■		■	■					■		▲			■	■	12
11	*Mycosphaerella gibsonii*	Needle blight of pine																															▲			1
12	*Adoxophyes orana*	Summer fruit tortrix moth		▲								■		■		■					▲	■	■	■				■	■					■	■	12
13	*Ceroplastes japonicus*	Japanese wax scale		■								▲									▲		■												■	■	6
14	*Oxycarenus hyalinipennis*	Cotton seed bug											■	■		■							■												■	■	6
15	*Agrilus biguttatus*	Oak splendor beetle																																	■	▲	2
16	*Platypus quercivorus*	Oak ambrosia beetle																																	■	▲	2
17	*Meloidogyne fallax*	False Columbia root-knot nematode			■		■			▲			■					■				■		▲					■		■						9
18	*Meloidogyne artiellia*	British root-knot nematode				■	■	■			■								■							■					■						7
19	*Ditylenchus angustus*	Rice stem nematode																							▲												1
20	*Heterodera latipons*	Mediterranean cereal cyst nematode				■				■									■			■									■						5
21	*Cronartium flaccidum*	Scots pine blister rust																															▲				1
22	*Phytophthora quercina*	Oak decline																																		■	1
23	*Lymantria mathura*	Pink gypsy moth		■																			■										■	■	▲	▲	6
24	*Cochlicella* spp.	Exotic species				■				■																					■		■	■	■	6
25	*Monacha* spp. (*M. cantiana*, *M. syriaca*)																																			0
26	*Onopordum acaulon*	Horse thistle																																			0
27	*Tomicus destruens*	Pine shoot beetle																															▲				1
28	*Thaumatotibia leucotreta*	False codling moth					■					▲	▲	▲		■					▲			■		▲								■	▲	10
29	*Dendrolimus pini*	Pine-tree lappet																															▲	■			2
30	*Chilo suppresalis*	Asiatic rice borer											▲												▲	▲											3
31	*Leucoptera malifoliella*	Pear leaf blister moth		■																	■		■												■	■	5
32	*Monochamus sutor*	Small white-marmorated longhorned beetle																															■	■		■	3
33	*Candidatus* Phytoplasma australiense	Phytoplasma yellows					■		■						■	▲					■								▲					■	■	8
34	*Heterodera cajani*	Pigeonpea cyst nematode					▲																														1
35	*Ophiostoma longicollum*																																			■	1
36	*Monochamus saltuarius*	Japanese pine sawyer																															▲	▲			2
37	*Spodoptera littoralis*	Egyptian cottonworm		■	■		▲	▲	■	▲	■	▲	▲	▲	▲	▲	▲			▲		▲	■	▲	▲	▲	▲	■	▲	▲	▲	■	■	■	■	28
38	*Xanthomonas oryzae*	Bacterial leaf streak, bacterial blight																							▲												1
39	*Archips xylosteanus*	Variegated golden tortrix		■								■									■		■	■										■	■	■	8
40	*Heterodera sacchari*	Sugar cane cyst nematode																							▲												1
41	*Cernuella* spp.	Exotic species				■	■																				■		■		■	■	■	■	■	9
42	*Meloidogyne paranaensis*	Parana coffee root-knot nematode																												■					■	2
43	*Harpophora maydis*	Late wilt of corn											▲																								1
44	*Meloidogyne mingnanica*	Citrus root-knot nematode										▲																									1
45	*Meloidogyne fujianensis*	Asian citrus root-knot nematode										▲																									1
46	*Phytophthora alni*	Alder root rot																																	▲		1
47	*Meloidogyne indica*	Citrus root-knot nematode										■																									1
48	*Meloidogyne citri*	Asian citrus root-knot nematode										■												■							■						3
49	*Meloidogyne donghaiensis*	Donghai root-knot nematode										▲																									1
50	*Meloidogyne jianyangensis*	Citrus root-knot nematode										■																									1

Version 9.4

3/27/2008

AHP Prioritized Pest List for FY09 - Prioritized

Rank	Scientific Name	Common Name	Almonds (*Prunus dulcis*)	Apples (*Malus* spp.)	Asparagus (*Asparagus* spp.)	Barley (*Hordeum* spp.)	Beans (*Phaseolus* spp.)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Carrots (*Daucus carota*)	Celery (*Apium graveolens*)	Citrus (*Citrus* spp.)	Corn (*Zea* spp.)	Cotton (*Gossypium* spp.)	Cucumbers (*Cucumis* spp.)	Grapes (*Vitis* spp.)	Lettuce (*Lactuca* spp.)	Oats (*Avena* spp.)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Peanuts (*Arachis* spp.)	Pears (*Pyrus* spp.)	Potatoes (*Solanum* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Soybeans (*Glycine* spp.)	Strawberries (*Fragaria* spp.)	Sunflower (*Helianthus* spp.)	Tomatoes (*Lycopersicon* spp.)	Wheat (*Triticum* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
51	*Meloidogyne kongi*	Citrus root-knot nematode										▲																							1
52	*Protopulvinaria longivalvata*											■																						■		2
53	*Dendroctonus micans*	Great spruce bark beetle																													■	▲			2
54	*Pulvinaria polygonata*	Cottony citrus scale										■																							1
55	Torradovirus	ToTV, ToANV, ToMarV																												▲					1
56	*Ostrinia furnacalis*	Asian corn borer					■						▲	■										■		■							■		6
57	*Scolytus intricatus*	European oak bark beetle																															■	▲	2
58	*Hylobius abietis*	Large pine weevil																													▲	▲		■	3
59	*Tropilaelaps clareae*	Bee mite																																	0
60	*Rathayibacter toxicus*	Annual ryegrass toxicity																																	0
61	*Pomacea* spp.	Exotic apple snails																							▲										1
62	*Monochamus alternatus*	Japanese pine sawyer beetle		■																											▲	■		■	4
63	*Cecidophyopsis ribis*	Black currant gall mite																																	0
64	*Crocidolomia binotalis*	Cabbage cluster caterpillar					■																												1
65	*Chilecomadia valdiviana*	Carpenter worm		■																			■										■	■	4
66	*Autographa gamma*	Silver Y moth		■		■	■	■	■	▲	■		▲	▲	■	▲	▲	■	■				■			▲		■	■	▲			■	■	21
67	*Phellinus noxius*	Brown root rot										■				■	■			■		■									■	■	■	■	9
68	*Stenchaetothrips biformis*	Rice thrips											■												▲										2
69	*Planococcus lilacinus*	Coffee mealybug								■	▲					■					■		■										■	■	7
70	*Rhabdoscelus obscurus*	Sugar cane weevil											■																				■		2
71	*Eutetranychus orientalis*	Citrus brown mite	■	■			■		■			▲	■	▲	■	■					■		■	■		■	■	■	■				■	■	■	19
72	*Synanthedon myopaeformis*	Red-belted clearwing		■																			■											■	■	4
73	*Hygromia cinctella*	Girdled snail						■																												1
74	*Chlorophorus strobilicola*	Pine cone cerambycid																													▲				1
75	*Elsinoe batatas*	Stem and leaf scab																																	0
76	*Gymnocoronis spilanthoides*	Senegal tea plant																							■										1
77	*Aleurocanthus spiniferus*	Orange spiny whitefly										▲				■					■		■										■	■	6
78	*Phoma tracheiphila*	Mal secco										▲																							1
79	*Pericyma cruegeri*	Poinciana looper																																■	1
80	*Ageratina riparia*	Mistflower																																	0
81	*Actinoscirpus grossus*	Giant bulrush																							■										1
82	*Rhynchophorus palmarum*	Palm Weevil										■																						■	2
83	*Arion vulgaris (Arion lusitanicus)*	Spanish slug				■	▲		■	■			■				▲		■					■				■	■						10
84	*Copitarsia* spp.	Copitarsia moths		■	■			■		■			■			■	■		■					■				■	■	■	■				■	14
85	*Metamasius* spp.	Exotic metamasius weevils											■													■									2
86	*Philephedra broadwayi*																																		0
87	*Coconut cadang-cadang viroid*	CCCVd																																	0
88	*Acroceras zizanioides*	Oat grass																							■						■	■	■	■	5
89	*Rubus alceifolius*	Giant bramble																													■	■	■	■	4
90	*Toxoptera odinae*	Mango aphid																																	0
91	*Peronosclerospora maydis*	Downy mildew of corn											▲													■									2
92	*Peronosclerospora philippinensis*	Philippine downy mildew											▲						▲							■									3
93	*Cataenococcus hispidus*	Cocoa mealybug										▲																					■	■	3
94	*Pinus roxburghii*	Chir pine																													■	■			2
95	*Heterodera ciceri*	Chickpea cyst nematode					■																				■								2
96	*Darna pallivitta*	Nettle caterpillar											■																						1
97	*Heteronychus arator*	Black maize beetle					■	■		■			▲			▲	■							▲		■		■		■	■				▲	12
98	*Lobesia botrana*	European grape vine moth				■										▲					▲		■	■										■	▲	7
99	Colocasia bobone disease virus	CBDV																																	0
100	*Juncus prismatocarpus*	Jointed rush																																	0

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AHP Prioritized Pest List for FY09 - Prioritized

Rank	Scientific Name	Common Name	Almonds (*Prunus dulcis*)	Apples (*Malus* spp.)	Asparagus (*Asparagus* spp.)	Barley (*Hordeum* spp.)	Beans (*Phaseolus* spp.)	Broccoli (*Brassica oleracea*)	Cantaloupes (*Cucumis* spp.)	Carrots (*Daucus carota*)	Celery (*Apium graveolens*)	Citrus (*Citrus* spp.)	Corn (*Zea* spp.)	Cotton (*Gossypium* spp.)	Cucumbers (*Cucumis* spp.)	Grapes (*Vitis* spp.)	Lettuce (*Lactuca* spp.)	Oats (*Avena* spp.)	Onions (*Allium* spp.)	Peaches (*Prunus persica*)	Peanuts (*Arachis* spp.)	Pears (*Pyrus* spp.)	Potatoes (*Solanum* spp.)	Rice (*Oryza* spp.)	Sorghum (*Sorghum* spp.)	Soybeans (*Glycine* spp.)	Strawberries (*Fragaria* spp.)	Sunflower (*Helianthus* spp.)	Tomatoes (*Lycopersicon* spp.)	Wheat (*Triticum* spp.)	Pine (*Pinus* spp.)	Other Softwood Trees*	Soft Hardwood Trees*	Hardwood Trees*	Pest Commodity Total
101	*Cydia funebrana*	Plum fruit moth	■	■																		■											■	■	6
102	*Sclerophthora rayssiae* var. *zeae*	Brown stripe downy mildew											▲								▲				▲										2
103	*Withania somnifera*	Ashwagandha										■																							1
104	*Synchytrium endobioticum*	Potato wart																						▲						■					2
105	*Meloidogyne mali*	Apple root-knot nematode		■												■																	■	■	4
106	Taro bacilliform virus	TaBV																																	0
107	*Meloidogyne coffeicola*	Coffee root-knot nematode																																	0
108	*Bursaphelenchus cocophilus*	Red ring nematode																															■		1
		Total Pests Per Commodity:	2	19	5	8	18	13	9	11	7	29	23	12	9	18	11	6	7	14	10	19	22	14	14	10	13	11	15	11	19	20	40	41	

Legend:

▲ = Primary host

■ = Other host

***Key to Forest Product Categories:**

Other Softwood Trees (Genera: *Abies, Casuarina, Chamaecyparis, Cupressus, Juniperus, Larix, Picea, Pseudotsuga, Sequoia, Taxus, Thuja, Torreya, Tsuga*)

Soft Hardwood Trees (Genera: *Acacia, Aesculus, Albizia, Alnus, Castanea, Catalpa, Celtis, Elaeagnus, Fraxinus, Gordonia, Liquidambar, Liriodendron, Magnolia, Melaleuca, Persea, Platanus, Populus, Sabal, Salix, Sassafras, Tamarix, Tilia, Ulmus*)

Hardwood Trees (Genera: *Acer, Ailanthus, Arbutus, Betula, Bumelia, Carpinus, Carya, Castanopsis, Cornus, Crataegus, Diospyros, Eucalyptus, Fagus, Ilex, Juglans, Lithocarpus, Malus, Melia, Morus, Ostrya, Prosopis, Prunus, Quercus, Robinia, Sapium, Sorbus, Umbellularia, Vaccinium*)

Commodities in Decreasing Order of Value:**

Corn (*Zea* spp.), Soybeans (*Glycine* spp.), Wheat (*Triticum* spp.), Cotton (*Gossypium* spp.), Tomatoes (*Lycopersicon* spp.), Grapes (*Vitis* spp.), Potatoes (*Solanum* spp.), Apples (*Malus* spp.), Citrus (*Citrus* spp.), Peanuts (*Arachis* spp.), Lettuce (*Lactuca* spp.), Rice (*Oryza* spp.), Sorghum (*Sorghum* spp.), Barley (*Hordeum* spp.), Strawberries (*Fragaria* spp.), Almonds (*Prunus dulcis*), Onions (*Allium* spp.), Peaches (*Prunus persica*), Carrots (*Daucus carota*), Cucumbers (*Cucumis* spp.), Beans (*Phaseolus* spp.), Sunflower (*Helianthus* spp.), Pears (*Pyrus* spp.), Celery (*Apium graveolens*), Broccoli (*Brassica oleracea*), Cantaloups (*Cucumis* spp.), Oats (*Avena* spp.), Asparagus (*Asparagus* spp.).
Not included in the ranking by value: Pine (*Pinus* spp.), Other Softwood trees*, Soft Hardwood trees*, Hardwood Trees*.

**

NASS. 2007. Agricultural Prices 2006 Summary. Agricultural Statistics Board, National Agricultural Statistics Service, United States Department of Agriculture.

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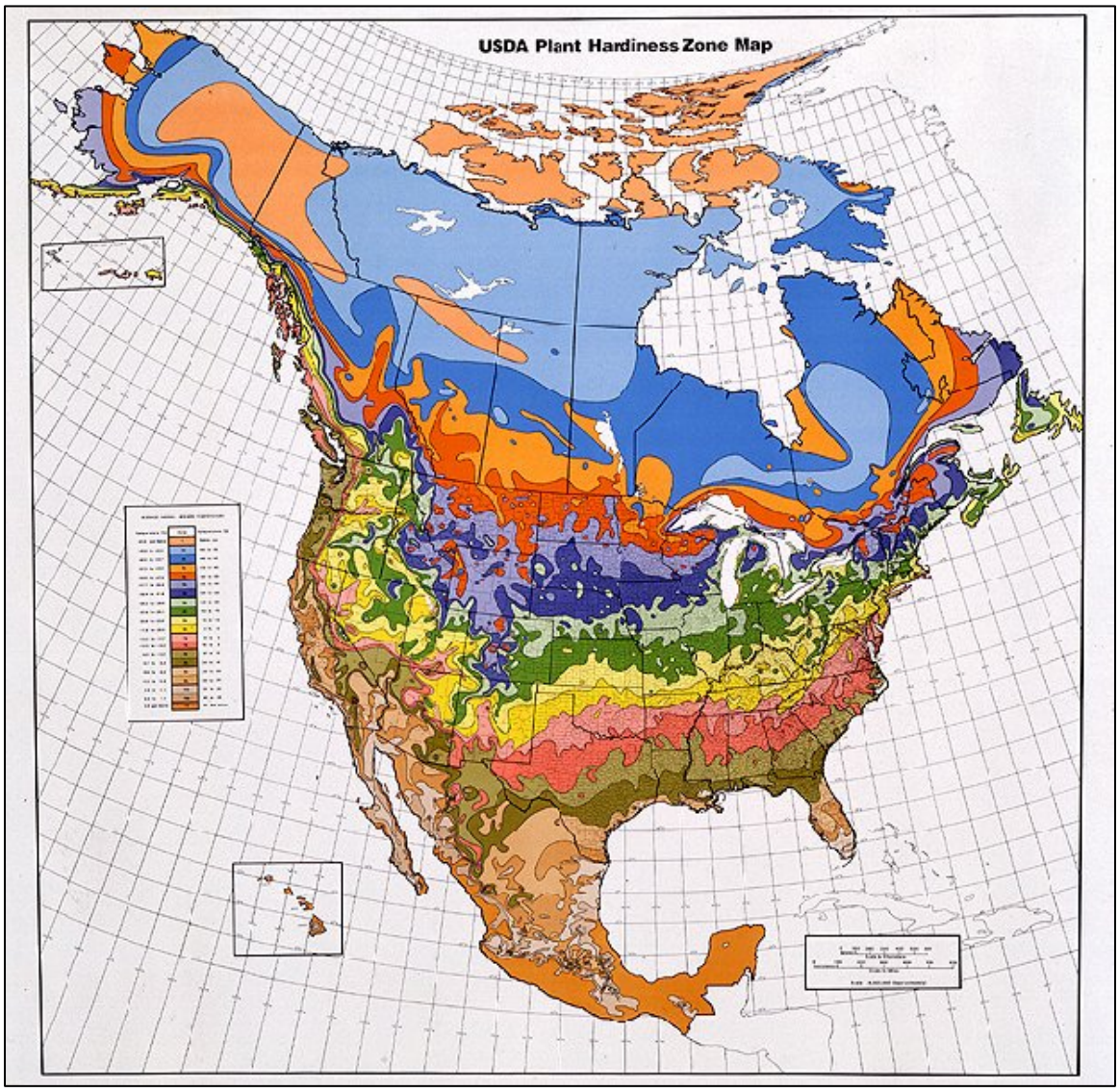
Appendix E: USDA Plant Hardiness Zone Map

(see http://www.usna.usda.gov/Hardzone/ushzmap.html for interactive map)

The preceding map was produced in 1990 by the US Department of Agriculture (USDA). This version shows in detail the lowest temperatures that can be expected each year in the United States, Canada, and Mexico.

These temperatures are referred to as "average annual minimum temperatures" and are based on the lowest temperatures recorded for each of the years 1974 to 1986 in the United States and Canada and 1971 to 1984 in Mexico. The map shows 10 different zones, each of which represents an area of winter hardiness for the plants of agriculture and our natural landscape. It also introduces zone 11 to represent areas that have average annual minimum temperatures above 40 F (4.4 C) and that are, therefore, essentially frost free. Actual temperature ranges for each zone are given below. Zones 2-10 in the map have been subdivided into light- and dark-colored sections (a and b) that represent 5 F (2.8 C) differences within the 10 F (5.6 C) zone.


USDA Plant Hardiness Zone Map

Zone	Fahrenheit	Celsius
1	Below -50 F	Below -45.6 C
2a	-50 to -45 F	-42.8 to -45.5 C
2b	-45 to -40 F	-40.0 to -42.7 C
3a	-40 to -35 F	-37.3 to -39.9 C
3b	-35 to -30 F	-34.5 to -37.2 C
4a	-30 to -25 F	-31.7 to -34.4 C
4b	-25 to -20 F	-28.9 to -31.6 C
5a	-20 to -15 F	-26.2 to -28.8 C
5b	-15 to -10 F	-23.4 to -26.1 C
6a	-10 to -5 F	-20.6 to -23.3 C
6b	-5 to 0 F	-17.8 to -20.5 C
7a	0 to 5 F	-15.0 to -17.7 C
7b	5 to 10 F	-12.3 to -14.9 C
8a	10 to 15 F	-9.5 to -12.2 C
8b	15 to 20 F	-6.7 to -9.4 C
9a	20 to 25 F	-3.9 to -6.6 C
9b	25 to 30 F	-1.2 to -3.8 C
10a	30 to 35 F	1.6 to -1.1 C
10b	35 to 40 F	4.4 to 1.7 C
11	above 40 F	above 4.5 C

Appendix F: 2008 Exotic Pest Detection Survey Order Form

2008 EXOTIC PEST DETECTION SURVEY ORDER FORM

Please fill out an order form for each state in which traps will be placed. States with more than one "ship-to" address should fill out a separate order form for each address.

If there is any trapping to be done within a state, an order form must be submitted to Otis **even if that state is not ordering any new lures**. Simply fill out the "number traps to be placed" column and put a "0" in the "number dispensers requested" column. This will provide the information to compile the annual report.

Lures for Khapra Beetle (*Trogoderma granarium*), European Cherry Fruit Fly (*Rhagoletis cerasi*) and Swede midge (Contarinia nasturtii) are to be purchased from commercial sources, but again, please indicate on the order form any trapping to be done for these species. All other lures are provided by the Otis Pest Survey, Detection and Exclusion Laboratory. Traps and liners are to be purchased locally or regionally.

If you have any questions about exotic pest survey trapping, please consult your **Exotic Pest Detection Manual**, and if there are remaining questions, please call or email Natalie Leva.

All orders for pheromone dispensers should be sent to:

Natalie Leva
USDA, APHIS, PPQ
Otis Pest Survey, Detection and Exclusion Lab
Building 1398
Otis ANGB, MA 02542

Tel: 508-563-9303 x 255
Fax: 508-564-4398
E-mail: natalie.m.leva@aphis.usda.gov

All orders should be received in the Otis lab by January 25, 2008.

Please use a Street Address, not a P.O. Box, for the Ship to Address and make sure you fill out all * fields, thank you.

State in which trap will be placed:	Date order placed:

Name: _____

Organization: _____

Address 1 _____

Address 2 _____

Phone _____

Email _____

Ship to Address

*Name: _____

*Organization _____

*Address 1 _____

Address 2 _____

*Phone _____

Email _____

Requested Ship Date: _____

Other Requests: _____

THE OTIS LAB DOES NOT SUPPLY ANY TRAPS OR TRAP PARTS

FOR OTIS USE ONLY:	
Shipment Date:	
Shipment Date:	☐
Shipment Complete:	☐

*Name: _____ *State: _____

Common Name / Species	Code	# Traps to be Placed	# Traps Ordered	# Trap Liners Ordered	Pheromone Dispenser Type	Frequency of Dispenser Replacement	# Dispensers Requested (Lures)
Leek Moth *Acrolepiopsis assectella*	LEM				Rubber septa	2 weeks	
Summer fruit tortrix moth *Adoxophyes orana*	ADOX				Rubber septa	12 weeks	
Apple Tortrix *Archips fuscocupreanus*	AF				Rubber septa	4 weeks	
Silver Y moth *Autographa gamma*	AG				Rubber septa	4 weeks	
Peach Fruit Moth *Carposina niponensis*	CN				Polycap	4 weeks	
Asiatic rice borer *Chilo suppressalis*	CS				Rubber septa	4 weeks	
Maize borer *Chilo partellus*	CP				Rubber septa	4 weeks	
Tomato Looper *Chrysodeixis chalcites*	TL				Rubber septa	4 weeks	
False codling moth *Cryptophlebia leucotreta*	FCM				Rubber septa	8 weeks	
Plum fruit moth *Cydia funebrana*	PFM				Rubber septa	4 weeks	
**Siberian Moth *Dendrolimus superans sibiricus*	SM				Rubber septa	4 weeks	
Cherry bark tortrix *Enarmonia formosana*	CBT				Rubber septa	4 weeks	
Light brown apple moth *Epiphyas postvittana*	LBAM				Rubber septa	4 weeks	
European grape berry moth *Eupoecilia ambiguella*	EA				Rubber septa	6 weeks	
Old World Bollworm *Helicoverpa armigera*	HA				Rubber septa	4 weeks	
Pear leaf blister moth *Leucoptera malifoliella*	PLBM				Rubber septa	10 weeks	
Grape vine moth *Lobesia botrana*	LB				Rubber septa	3 weeks	
*Rosvy moth *Lymantria mathura*	RM				String	12 weeks	
Nun moth *Lymantria monacha*	NM				Laminate	12 weeks	
Cabbage moth *Mamestra brassicae*	MB				Polycap	12 weeks	
(No Common Name) *Pandemis heperana*	PH				Rubber septa	4 weeks	
European cherry fruit fly *Rhagoletis cerasi*	RC				Polycap	2 weeks	OTIS DOES NOT SUPPLY
Egyptian cottonworm *Spodoptera littoralis*	ECL				Laminate	12 weeks	
Rice cutworm (cotton leafworm) *Spodoptera litura*	CL				Laminate	12 weeks	
Khapra beetle *Trogoderma granarium*	KB				Rubber septa	4 weeks	OTIS DOES NOT SUPPLY
Cherry ermine moth *Yponomeuta padellus*	CEM				Rubber septa	12 weeks	
Apple ermine moth *Yponomeuta malinellus*	AEM				Red Rubber septa	12 weeks	
***Swede midge *Contarinia nasturtii*	SWM				Polycap	12 weeks	OTIS DOES NOT SUPPLY

*Pending Lure Availability
***Jackson Trap

**Requires modified milk carton GM traps, modification available upon request

2008 Exotic Pest Order Form