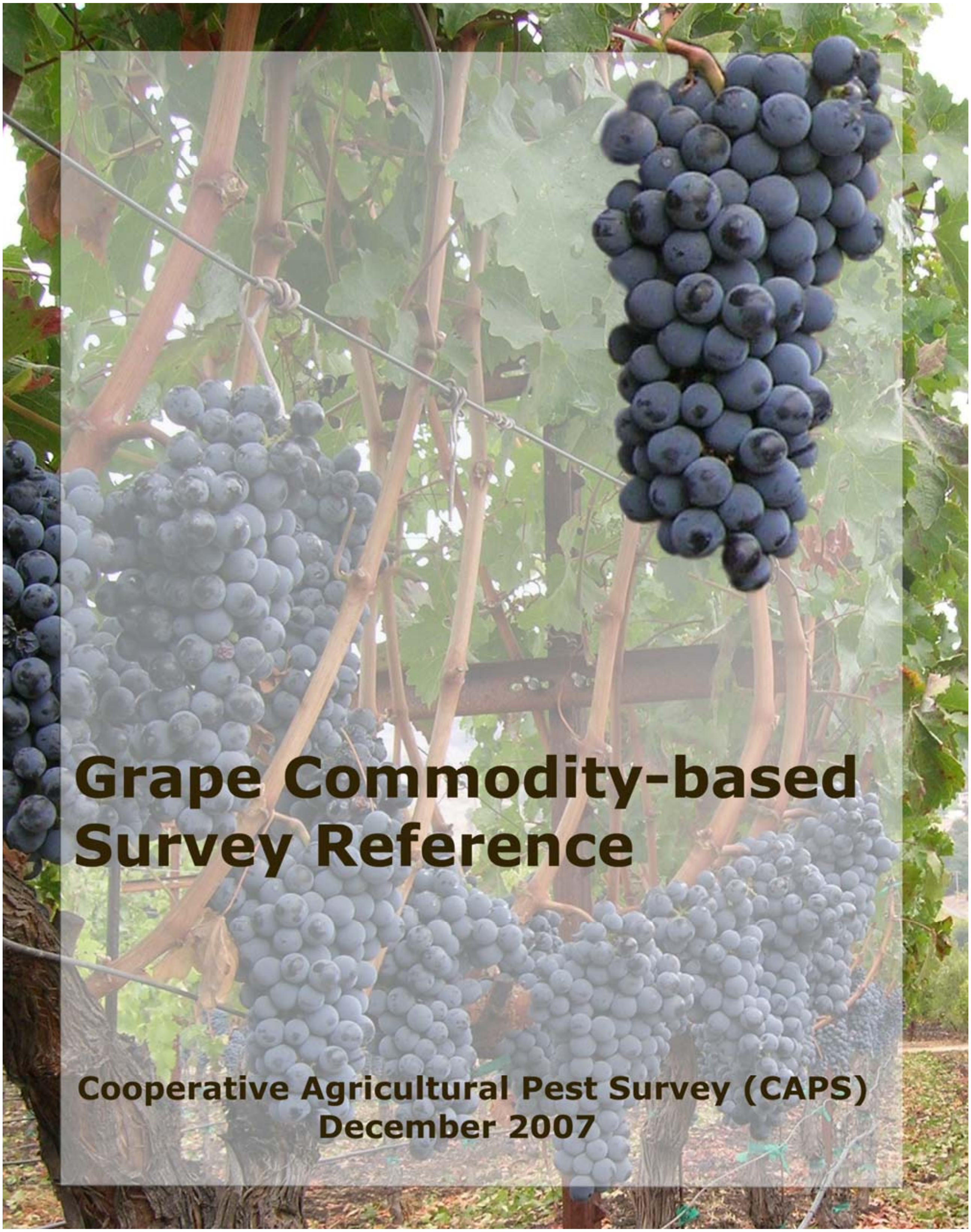

Grape Commodity-based Survey Reference

Cooperative Agricultural Pest Survey (CAPS)
December 2007

Table of Contents

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Introduction to Reference

History of Commodity-Based Survey

The 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 (in some cases) diagnose exotic and invasive plant pests. By finding pests early, eradication efforts will likely be less expensive and more efficient. For more information on 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 choose (from this list) 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 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 AHP prioritized pest list [http://ceris.purdue.edu/caps/adm2008/adm2008000019.pdf (this is a restricted site; password required) 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 AHP prioritized pest list for FY 08' and 09' are also given in Appendices C and D. 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. 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, small grains, and oak forests.

Grape Commodity Survey Reference

The *Grape Commodity Survey Reference* (CSR) is a companion document to the *Grape Commodity Survey Guidelines* (CSG). Both documents are intended to be tools to help survey professionals develop surveys for exotic grape pests. The *Grape CSR* is a collection of detailed data sheets on exotic pests of grape. Additionally, the authors have tried to identify native pests that these exotic pests may be easily confused with 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. In contrast, the *Grape Commodity Survey Guidelines* 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 guidelines contain little information about biology. Instead, they focus on survey design, sampling strategies, and methods of identification. There is no 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 guidelines 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 Grape 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 survey committee in more than one state.

State Survey Coordinators (SSC): The SSC is a state employee responsible for coordinating 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 PPQ 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 Grape Survey Reference

Organisms included in the grape 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 grape [*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 grape.

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.

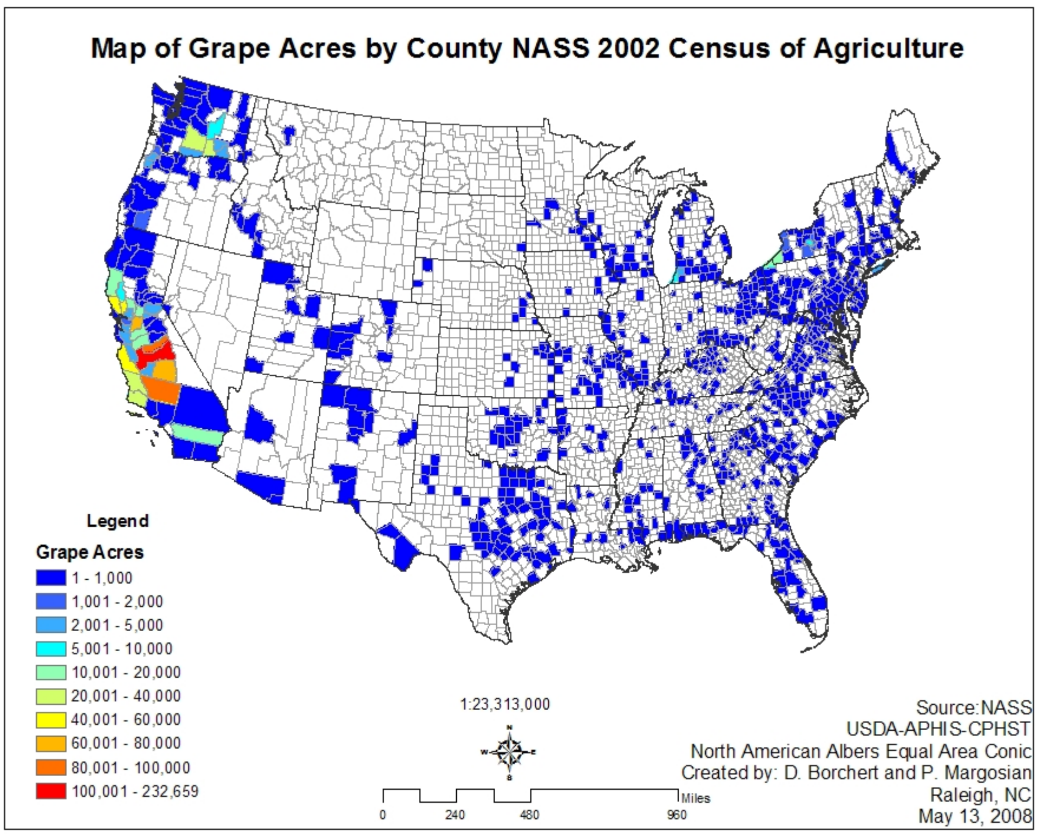
To help provide a rationale for the inclusion of each pest in the reference, the authors have included a section titled, “Reason for Inclusion in Manual”. Pests are either considered to be a CAPS target and are listed in the CAPS prioritized pest list or a national threat. The pests considered as national 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. After review from CAPS cooperators an additional reason for inclusion in the manual was included, “At the request of CAPS Cooperators”.

Introduction to Grape

Cultivated grapes, consumed as fresh fruit, raisins, wine, and juice, are an important crop in the United States. The many varieties and cultivars of grape belong to the *Vitaceae* family. The *Vitis* genus grows in eastern Asia, Europe, the Middle East and North America between 25° and 50° N latitude. Grapes can be classified by either food usage or by species (Reiger, 2006).

Grape production in the United States

The United States ranks fourth worldwide in grape production, contributing 8% of the total pounds produced globally. The U.S. grape industry is valued at approximately $3.1 billion dollars. In 2006, the United States produced a total of 6.33 million tons of grapes, valued at $499 per ton. Grapes are the highest value non-citrus fruit/nut crop in terms of both utilized production and value. Grape-producing states include Arizona, Arkansas, California, Georgia, Michigan, Missouri, New York, North Carolina, Ohio, Oregon, Pennsylvania, Texas, Virginia, and Washington. The majority of U.S. grapes, 90%, are grown in California with 800,000 acres in production (NASS, 2007).



Species and Cultivars

The genus *Vitis* is divided into two subgenera: *Euvitis* and *Muscadinia*. *Euvitis*, or 'true grapes,' are characterized by fruits growing in elongated clusters and attached to stems at maturity. *Euvitis* grapes have forked tendrils, diaphragms in pith at nodes, and long strips of loose bark detached from the vine. *Muscadinia*, or Muscadine grapes, have small clusters of thick-skinned fruit which detach as they mature. Tendrils of Muscadine grapes are simple, diaphragms in pith at nodes are lacking, and vines have smooth bark with lenticels (Reiger, 2006).

Grape production worldwide primarily consists of only four species or hybrids. *Vitis vinifera* L., often named the 'Old World grape,' is prized for use in wine production and for table and raisin grapes. Worldwide production is dominated by *V. vinifera*, with 90% of grape production in this species with at least 500 *V. vinifera* cultivars grown. *Vitis rotundifolia* Michx is a muscadine grape that is disease tolerant and grows vigorously. This species is not cold hardy but is well adapted to the southeastern United States. *Vitis labrusca* L. includes the popular 'Concord' cultivar, among others, and is grown for grape juice, jams, and associated products. Lastly, French-American hybrids between vinifera grape and native species rootstocks are grown to provide increased *Phylloxera* resistance (Reiger, 2006).

Food Usage

Classification of grapes by food usage includes table grapes, raisin grapes, sweet juice grapes, and wine grapes. Table grapes are those grown for and consumed as fresh fruit. *V. vinifera* 'Thompson seedless' grapes are widely grown as table grapes, as well as many other cultivars. 'Thompson seedless' is the most common raisin grape cultivar, making up 90% of raisin production in the United States. Sweet juice grapes grown for juice, jelly, jam, preserves and certain types of wine are typically 'Concord' grapes. Commercial wine grape production is dominated by cultivars of *V. vinifera* (Reiger, 2006).

Domestication

It is thought that grapes, *Vitis vinifera*, are native to southwestern Asia near the Caspian Sea. Like other fruits native to that region, such as pears and apples, grapes most likely spread to new places as a result of trade. Grape seeds dating back to the Bronze Age have been found in excavated dwellings in south-central Europe. Egyptian hieroglyphics told of wine making. Grapes were probably brought to Greece, Rome and France by the Phoenicians. Early settlers to North America brought grape starts, but the grapes faired poorly on the east coast. Spanish missionaries introduced vinifera-type grapes to California in the 1700s, where they have flourished as a crop (Reiger, 2006).

Biology and Reproduction of Grapes

All grapes are woody, climbing vines. Muscadine grapes have smooth bark with lenticels and small, round, unlobed leaves with dentate margins. Vinifera grapes are characterized by older vines with loose, flakey bark and large leaves. Vinifera leaves can vary greatly in both size and shape and may or may not be lobed. Small, green flowers appear on racemose panicles at the base of the current season's growth. Flowers have five sepals, five petals and five stamens. Superior ovaries have two locules, each with two ovules. Vinifera and Concord grapes have perfect flowered, self-pollinating flowers, and some muscadine cultivars are only found with pistillate flowers. The majority of grapes are self-pollinating, with some exceptions in muscadine grapes. Fruits of grapes are true berries, round to oblong in shape, and have up to four seeds. Skin is variable in thickness, but most is thin. A fine, glaucous layer of wax may be present on the fruit surface. Anthocyanin compounds are found in the skin, which colors the fruit red, blue, purple, or black (Reiger, 2006).

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Arthropods

Primary Pests of Grape (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

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

Pest Description
Eggs: Semi-spherical, 0.57 mm in diameter. Eggs are yellowish-white (Fig. 1A), 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.

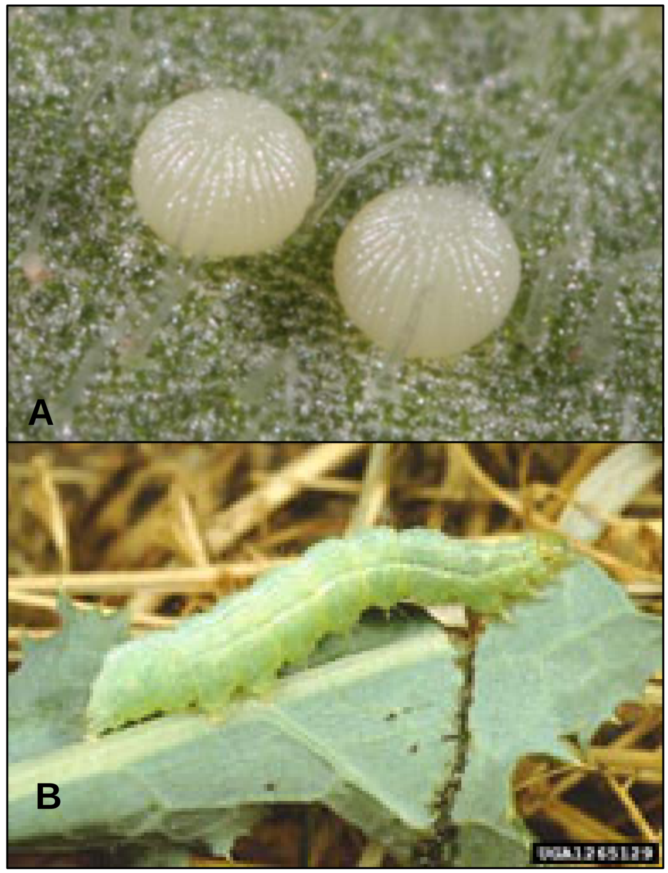

Figure 1. Eggs (A) and larvae (B) of *A. gamma* (A.) and . Photos courtesy of Jurgen Rodeland and P. Mazzei (www.invasive.org), respectively.

Larvae: The larva is a semi-looper with three pairs of prolegs. It occurs in varying shades of green (Fig. 1B), 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 is 20 to 40 mm (Emmett, 1980; Jones and Jones, 1984; USDA, 1986; Hill, 1983).

Pupae: Pupation takes place within a translucent, whitish cocoon spun amongst plant foliage (Fig. 2A). 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, and 4.5 to 6.0 mm broad. Cremaster globular, with four pairs of hooklets (Paulian et al., 1975; Carter and Hargreaves, 1986).

Adult: The adults are grey-colored and the forewings are marbled in appearance; their color being silvery-grey to reddish-grey to black with a velvety sheen. Wing expanse is 36 to 40 mm. The 'Y' mark on the forewing is distinct and silvery (Fig. 2B). The hindwings are brownish with a darker border (USDA, 1958; Jones and Jones, 1984; Hill, 1983).

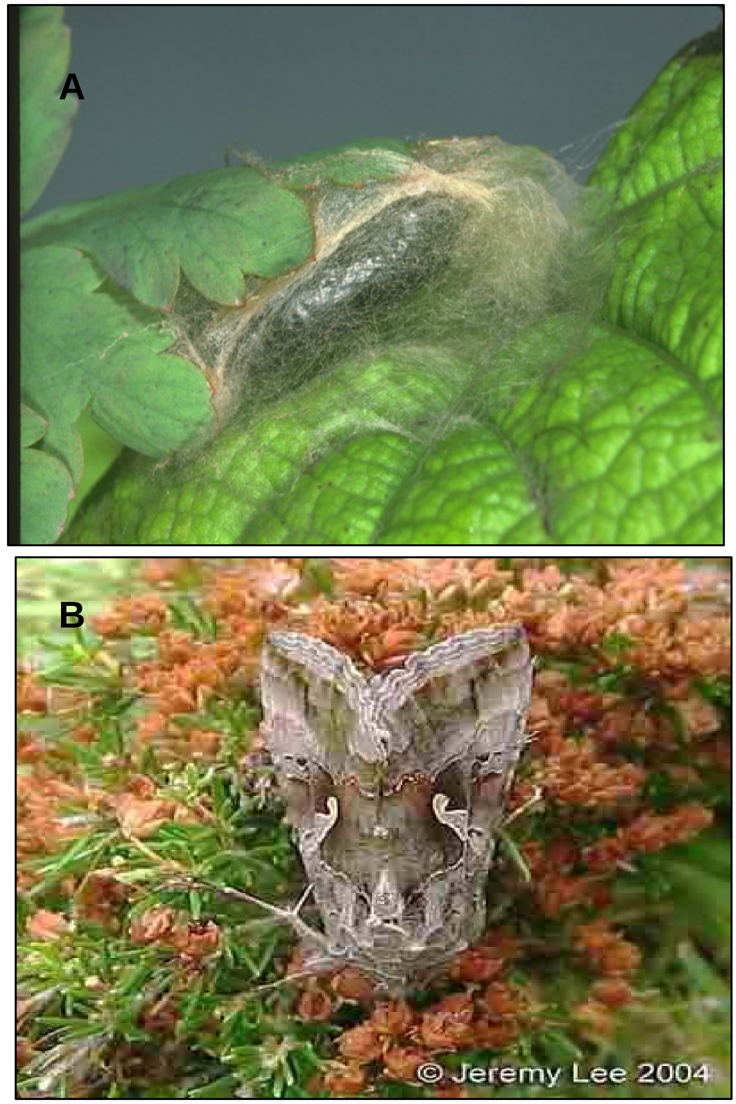

Figure 2. Cocoon (A) and adult (B) *A. gamma*. Photos courtesy of Alain Fraval and Jeremy Lee, respectively.

Biology and Ecology

A. gamma is a migratory species and adults undertake seasonal migrations to areas where they are able to breed. The silver-Y moth can be found in many habitats including agricultural land, waste land, and gardens. In areas where *A. gamma* is unable to overwinter, severe infestations occur sporadically.

Female moths take nectar from flowers and can often be seen feeding during the day or early evening. Females lay from 500 to more than 1000 whitish eggs (Hill, 1983) (Fig. 1A), singly or in small batches, on the underside of leaves of low-growing plants. In temperate regions, hatching may take 10 to 12 days (Hill, 1983). The incubation period lasts for 3 days at 25°C (Ugur, 1995).

The young larvae feed on the foliage of their host plants and tend to occur singly, rather than in groups. When they are young, they skeletonize the leaves, but older caterpillars eat the whole leaf (Hill, 1983). 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). When the larvae are disturbed, they drop off the plant.

Local distribution, reproductive potential and migration are determined to a considerable extent by the availability of suitable wild plants in a given area, and good weed control reduces the threat of outbreaks. Mortality in the egg stage and the first larval instar is lowest at high humidities; mass outbreaks are known to have occurred mainly during periods of very wet weather (Maceljski and Balarin, 1974).

In areas where *A. gamma* is able to survive the winter, it overwinters as third to fourth larval instars (Tarabrina, 1970; Kaneko, 1993; Saito, 1988) or in the pupal stage (Dochkova, 1972). There is no true diapause (Tyshchenko and Gasanov, 1983).

Symptoms/Signs

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 petioles or leaf stalks may be cut by the larvae. Frass may or may not be visible. Apart from damaging the foliage of their host plants, larvae can scrape the skin from grapes and feed on the contents of the fruits. A single larva can damage 20 or more mature grapes (Abdullagatov and Abdullagatov, 1986). 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.

Pest Importance

From CABI (2004):
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* in areas of the Netherlands was valued at as much as 320,000 guilders during some years in the 1800s. It is also very destructive in England and Denmark. Damage to globe artichokes was severe near Bari, Italy in 1982 to 1985, with about 55% of plants being damaged. *A. gamma* was one of the major pests.

Studies in Czechoslovakia (Novak, 1975) indicated that damage became of economic significance when 25% of the leaf area of a plant was destroyed. The critical density of larvae was, therefore, the number of larvae/unit area required to do this, which varied according to both the larval instar concerned and the development stage of the plant. The numbers of larvae per plant causing 25% leaf loss varied from 0.07 when the plant had only two leaves to 20 when it had 30 leaves.

Known Hosts

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

Major hosts
Beta vulgaris (beet), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Borago officinalis* (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* (hemp), *Capsicum* spp. (peppers), *Chrysanthemum indicum* (chrysanthemum), *Cicer arietinum* (chickpea), *Cichorium intybus* (chicory), *Cynara scolymus* (artichoke), *Daucus carota* (carrot), *Glycine max* (soybean), *Gossypium* spp. (cotton), *Helianthus annuus* (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* (purple clover), *Triticum aestivum* (wheat), *Vitis vinifera* (grape), *Zea mays* (maize), and *Zinnia elegans* (zinnia)

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

Known Distribution
A. 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 to become 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). Venette et al. (2003) estimated that approximately 48% of the continental U.S. would be suitable for establishment of *A. gamma*

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 from 100:1 to 95:5 (19:1) 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 crop height and raised as the crop matures. **This lure is available from CPHST-Otis** (formally the OTIS Pest Survey, Detection, and Exclusion Laboratory) **in the 100:1 ratio.**

<u>Alternative Method:</u> 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).

<u>Not Recommended:</u> 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 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. 3)*, Syngrapha celsa* (Fig. 4)*, A. pseudogamma* (Fig. 5), and *A. californica* (alfalfa looper) (Fig. 6). All 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”. See Nazmi et al. (1981) for a comparison of similarities and differences between closely related species.


Figure 3. Adult and larva of *Trichoplusia ni*. Photos courtesy of Keith Naylor and Extension Entomology, Texas A&M University.


Figure 4. Adult and larva of *Syngrapha celsa*. Photos courtesy of John Cooper and Natural Resources Canada.

Figure 5. Adult of *Autographa pseudogamma.* Photo courtesy of Natural Resources Canada.

Figure 6. Adult and larva of *Autographa californicum.* Photos courtesy Franklin Dlott, UC Cooperative Extension, Monterey County.

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Copitarsia spp.

Scientific Names of Species of Concern
Copitarsia incommoda Walker
Copitarsia decolora Herrich-Schaffer

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

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).

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

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

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

Type of Pest
Moth

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

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

Pest Description

General (all species in genus): *Copitarsia* spp. begin life as eggs (Fig. 1A), deposited singly or in egg masses. 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 grey 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).

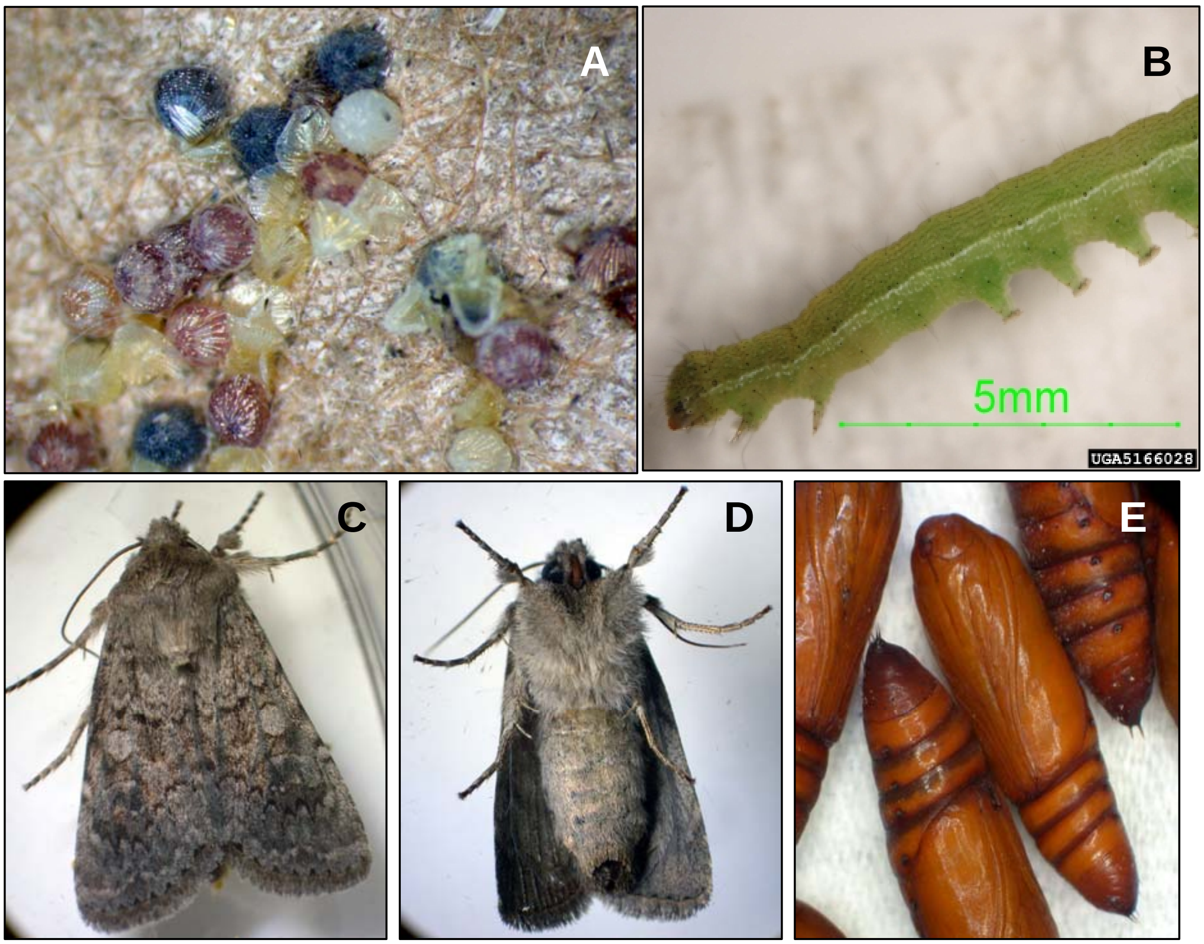

Figure 1. *Copitarsia* life stages. Eggs (A), larva (B), adult (C, D), and pupae (E). Photos courtesy of Julie Gould and Charles Olsen (USDA APHIS PPQ).

Copitarsia spp. pupate in the soil (Fig. 1E) and emerge as grey 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* spp. are multivoltine through much of their range. In general, *Copitarsia* spp. appear to have two to four generations per year.

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

Discussion. *C. decolora* varies slightly in coloration from light 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. *C. 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 grey; mesothorax pale brown; metathorax grey 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 grey; 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 greyish-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 grey, remainder of abdomen grey; genital tuft grey; 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. *C. incommoda* varies slightly in coloration from lighter to medium brown. Females tend to be larger and have darker hindwings than males.

Diagnosis. *C. 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 is 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 an 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 9.7^0 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 29.5^0 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 U.S. 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 annus* (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 pretense* (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* (husk tomato), *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. (baby's breath), *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 their 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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Epiphyas postvittana

Scientific Name
Epiphyas postvittana Walker

Synonyms:
Tortrix postvittana, Austrotortrix postvittana, Cacoecia postvittana, Teras postvittana, Archips postvittanus

Common Names
Light brown apple moth, apple leafroller, Australian leafroller

Type of Pest
Moth

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

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

Pest Description
Eggs: Pale green to pale brown (Fig. 1A), almost flat, 0.84 to 0.95 mm (USDA, 1984). Females deposit eggs in egg masses. Within a mass, eggs are "stuck together like roof tiles" and covered in a greenish "waxy secretion" (Evans, 1937; Geir and Briese, 1981).

Larvae: First instar larvae are approximately 1.6 mm long, and final instar larvae range from 10 to 20 mm in length. The body of a mature larva is green with a darker green central stripe and two side stripes (Fig. 1B). The first larval instar has a dark-brown head; all other instars have a light-fawn head and prothoracic plate. Overwintering larvae are typically darker (CABI, 2004).

Pupae: Pupae are green after pupation (Fig. 1C), but become brown within one day. Male pupae average 2.5 by 7.6 mm; females average 2.9 by 9.8 mm. The pupal stage is completed within the "nests" made up of rolled up leaves.

Adult: Light brown apple moth adults are highly sexually dimorphic (males are usually smaller) and variable in wing pattern and color, although a lighter, diamond-shaped area extending from behind the head to approximately one-third of body length is typically visible at rest (Fig. 1D) (CABI, 2004).. Male forewing length ranges from 6 to 10 mm, compared with 7 to 13 mm in females (Thomas, 1975). Males tend to have a higher contrast in coloration than females, although the level of contrast varies (Fig. 2). **Typical males have a light brown area at the base distinguishable from a much darker, red-brown area at the tip. The latter may be absent, the moth appearing uniformly**

light brown, as in the females, with only slightly darker oblique markings distinguishing the area at the tip of the wing.

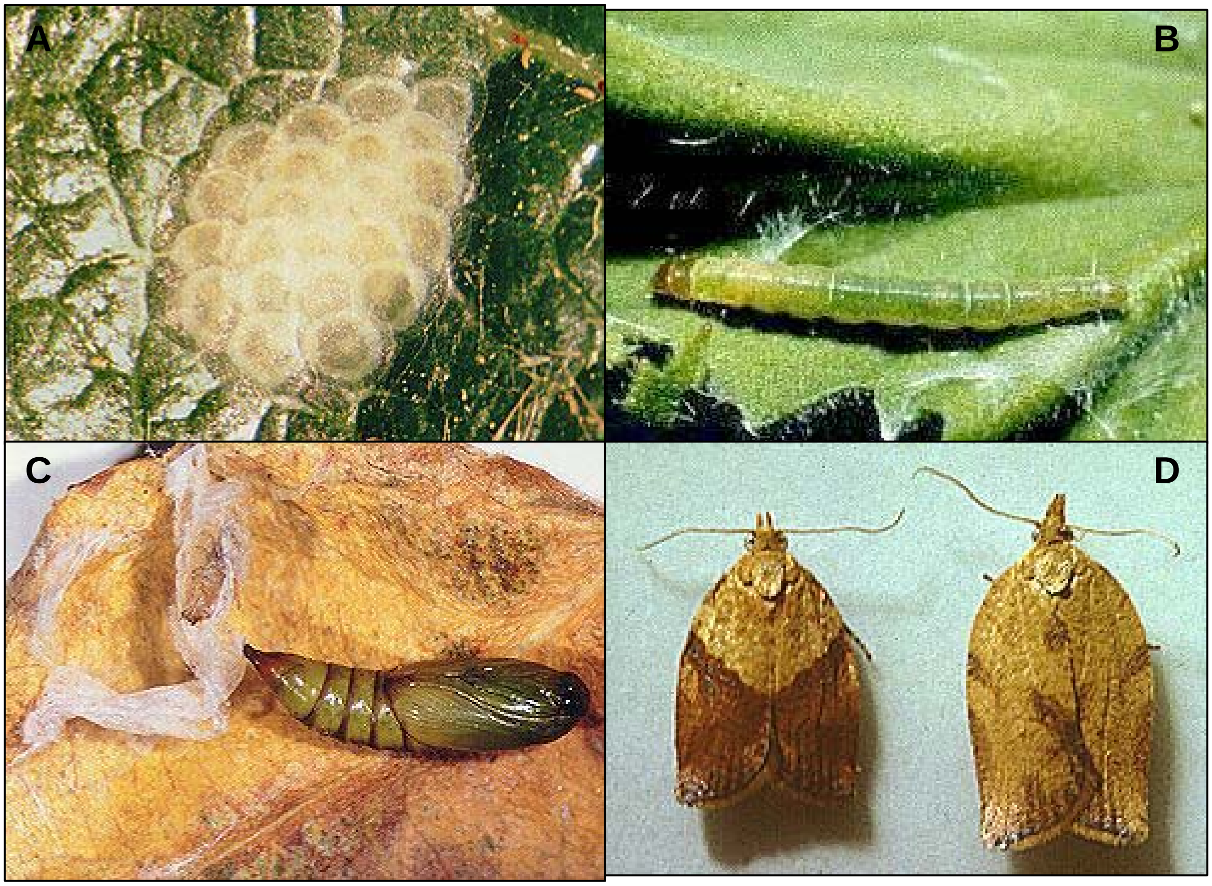

Figure 1. Life stages of *Epiphyas postvittana*: (A) eggs; (B) larva; (C) pupa, (D) adults, male is on the left. Photos courtesy of http://www.hortnet.co.nz/key/keys/info/lifecycl/lba-desc.htm

More detailed technical descriptions of the morphology of *E. postvittana* are provided by Zimmerman (1978), Hampson (1863), Bradley (1973), Bradley et al. (1979), and Scott (1984). Additional detail is also given in the Venette et al. (2003) mini-pest risk assessment at:
http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/epostvittanapra.pdf.

Biology and Ecology

The number of annual generations of light brown apple moth (LBAM) varies with latitude within its range. There is considerable overlap between generations, with development driven by temperature and larval host plant (Danthanarayana, 1975; Geier and Briese, 1980; Thomas, 1989). The highest rate of population increase was on *Plantago lanceolata* (ribwort plantain), followed by *Rumex crispus* (curly dock), apples (*Malus domestica* cv. Granny Smith) and *Trifolium repens* (white clover) (Danthanarayana et al., 1995). Cooler temperatures lead to longer development times for all stages of growth (Magarey et al., 1994). In summer, the life cycle takes 4 to 6 weeks to complete

Figure 2. *E. postvittana* (museum set specimen); adult male (A) and adult female (B). Photos courtesy of CABI, 2004.

(Nuttal, 1983), but more than three generations can be completed if temperatures and host plants are favorable (MacLellan, 1973; Thomas, 1989; Buchanan et al., 1991; Madge and Stirrat, 1991; Magarey et al., 1994; Bailey, 1997).

Adult moths emerge after one to several weeks of pupation. Female moths emerge from protective pupal nests and mate soon after emergence (Geier and Briese, 1981). Danthanarayana (1975) suggests the preoviposition period is 2 to 7 days. Females copulate for slightly less than 1 hr (Foster et al., 1995). Oviposition does not begin until females are 2- to 3-days old (Geier and Briese, 1981). The oviposition period lasts 1 to 21 days (Danthanarayana, 1975). Adult longevity is influenced by host plant and temperature. In the laboratory, female longevity can vary between 10 days (Geier and Briese, 1981) and 32.7 days (Danthanarayana, 1975); males can live up to approximately 33 days (Danthanarayana, 1975). Under field conditions in Australia, the life span of adult *E. postvittana* is 2 to 3 weeks (Magarey et al., 1994).

Moths are quiescent during the day and may be found on foliage of hosts (Geier and Briese, 1981). Long distance dispersal is typically achieved by adults (Geier and Briese, 1980; Suckling et al., 1994), although larval dispersal occurs over a short range. Flight occurs at dusk in calm conditions (Geier and Briese, 1981; USDA, 1984; Magarey et al., 1994). Females deposit eggs at night (USDA, 1984). Adults are unlikely to disperse from areas with abundant, high-quality hosts (Geier and Briese, 1981). Males will disperse farther than females. In a mark-release-recapture study, 80% of recaptured males and 99% of recaptured females occurred within 100 m of the release point (Suckling et al., 1994). Females do not appear to rely on plant volatiles to locate a host, but tactile cues are important (Foster and Howard, 1998). Females prefer smooth leaf surfaces on which to deposit their eggs (Danthanarayana, 1975; Geier and Briese, 1981; Foster and Howard, 1998). Humidity influences the dispersal ability of the pest (Danthanarayana et al., 1995).

Females deposit eggs in egg masses. The number of eggs deposited in a mass is variable. Typically, females deposit 20 to 50 eggs per mass. A female moth may produce up to 1,492 eggs (Danthanarayana 1975, 1983), but the average number of eggs produced per female typically varies from 118 to 462 (MacLellan, 1973; Danthanarayana, 1975; Geier and Briese, 1981; USDA, 1984; Danthanarayana et al., 1995). Fecundity is greatest at temperatures between 20 and 25°C [68 to 77°F], inclusive (Danthanarayana et al., 1995). The egg stage lasts an average of 5 to 7 days at a temperature of 28°C [82°F] (Danthanarayana, 1975). Egg-hatching ceases at temperatures greater than 31.3°C [88°F] (Danthanarayana, 1975).

E. postvittana typically completes five to seven instars (Danthanarayana, 1975; Geier and Briese, 1981; Magarey et al., 1994). Larvae emerge from eggs after 1 to 2 weeks and disperse, usually to the underside of the leaf, where they spin a “silken shelter” (*i.e.*, a silken tunnel) and commence feeding (Danthanarayana, 1975; Geier and Briese, 1981; Nuttal, 1983; USDA, 1984; Thomas, 1989). Although they are sheltered in silk, first instar larvae are more exposed to weather and insecticide treatments than are second and third instar larvae (Madge and Stirrat, 1991; Lo et al., 2000). After approximately 3 weeks, larvae leave the silken tunnels for a new leaf (USDA, 1984). Second and later instars have the ability to create their own protective feeding shelter by rolling a leaf or webbing multiple leaves together (Danthanarayana, 1975; Lo et al., 2000), behaviors characteristic of the Tortricidae.

Larvae move vigorously when disturbed, but are always connected to the leaf by a silken thread to avoid being removed from the leaf (Nuttal, 1983; USDA, 1984). When larvae happen to fall to the ground, they feed on ground-cover hosts or can survive without feeding for several months (Evans, 1937; Thomas, 1975; USDA, 1984).

In cold climates, *E. postivttana* overwinter in the larval stage (Nuttal, 1983). Larvae prepare to overwinter by locating “sheltering niches,” which may be mummified fruit or ground vegetation (Thomas, 1975). Overwintering larvae can utilize alternate hosts, including several weed species, for food and to form shelters (Buchanan et al., 1991). Larvae may also survive winters without feeding for up to 2 months (USDA, 1984). *E. postvittana* does not diapause (Geier and Briese, 1981); rather, development is slowed under cold winter temperatures (MacLellan, 1973; Geier and Briese, 1981; Danthanarayana, 1983; USDA, 1984). Development is only likely to occur at temperatures between 7.1° and 30.7°C [45-87°F] (Danthanarayana et al., 1995).

Pupation is completed within the “nests” made from rolled-up leaves (Danthanarayana, 1975; Geier and Briese, 1981; Nuttal, 1983; Magarey et al., 1994). The pupal stage lasts 2 to 3 weeks (Evans, 1937).

E. postvittana is more abundant during the second generation than during other generations (MacLellan, 1973; Madge and Stirrat, 1991). Thus, the second generation causes the most economic damage (Evans, 1937; Thomas, 1975; Madge and Stirrat, 1991; Lo et al., 2000) as larvae move from foliage to fruit (MacLellan, 1973; Magarey et al., 1994).

Symptoms/Signs

The insect will feed on foliage, flowers, and fruit. In spring, the pest feeds on new buds while later generations feed on ripened fruits (Buchanan et al., 1991). After the first molt, they construct typical leaf rolls (nests) by webbing together leaves, a bud and one or more leaves, leaves to a fruit, or by folding and webbing individual mature leaves. During the fruiting season, they also make nests among clusters of fruits, damaging the surface and sometimes tunneling into the fruits (Danthanarayana, 1975).

Fruit surface feeding is common within larval nest sites and is typically caused by later instars (Lo et al., 2000). Clusters of fruit are particularly susceptible. *E. postvittana* has been shown to introduce *Botrytis cinerea* spores into wounds via contaminated larvae, with up to 13% of berry damage (by weight) as a result (Bailey, 1997). On a fruit, the calyx offers protection from parasitoids and is probably the best feeding location for young larvae (Lo et al., 2000). Larvae entering the fruit through the calyx may cause internal damage. Wet conditions may allow the entry of rot organisms. Feeding on the foliage by larvae causes ragging and curling of the foliage.

Damage to apples is in the form of either pinpricks, which are flask-shaped holes about 3 mm deep into the fruit, or entries, which are holes extending deeper than 3 mm into the fruit that leaves some frass and webbing at the surface (van Den Broek, 1975). On apples, skin damage or blemishes have an irregular cork-like appearance. Larvae may excavate small round pits and produce scars similar to the "stings" of the larvae of *Cydia pomonella*, the codling moth. The first generation (in spring) causes the most damage to apples; while the second generation damages fruit harvested later in the season (Terauds, 1977). Peaches are damaged by feeding that occurs on the shoots and fruit.

Pest Importance

The larva of *E. postvittana* is a serious pest of fruit and ornamentals in Australia and New Zealand. As a pest of pome fruits, particularly apples, it probably ranks second to *Cydia pomonella*, the codling moth. During a severe outbreak, damage by *E. postvittana* to fruit may be as much as 75%. In Tasmania, this species is the most injurious pest of apples. In years of abundance, populations of the light brown apple moth may cause as much as 25% loss of the apple crop. This pest damages fruit in storage; a few larvae may ruin a whole case of fruit. The markings on the fruit render it unfit for export (USDA, 1984).

E. postvittana is a highly polyphagous pest that attacks a wide number of fruits, ornamentals, and other plants. According to Geier and Briese (1981), "Economic damage results from feeding by caterpillars, which may destroy, stunt or deform young seedlings, spoil the appearance of ornamental plants, and/or injure deciduous fruit-tree crops, citrus, and grapes." Losses in Australia were estimated to be AU$21M per annum, but there has been no similar estimation in other countries.

The larvae can be very damaging to grape, apple, and peach. In grape, 70,000 larvae/ha were documented to cause a loss of 4.7 tons of chardonnay fruit in 1992 with an estimated cost of $2000/ha (Bailey et al., 1995). A single larva can destroy about 30 grams of mature grapes.

Mature larvae are the most difficult stage to control. *E. postvittana* is also difficult to control with sprays because of its leaf-rolling ability, and because there is evidence of resistance due to overuse of sprays (Geier and Briese, 1981).

Canada has listed *E. postvittana* as a noxious pest, and the presence of the pest would prevent export of any infested commodity destined to Canada (Danthanarayana et al., 1995). In New Zealand, the recommended economic threshold is six or more larvae per 30 meter row of fruit crops; however, if the crop is intended for export, control is recommended if only one larva is found (Charles et al., 1987).

Known Hosts

In its native Australia, this species is thought to have evolved in association with *Acacia* spp.(wattles) and other evergreen species (Danthanarayana, 1975). *E. postvittana* has colonized a wide range of orchard and other habitats in both Australia and New Zealand. It is present in pine forests on understory perennial weeds, on willows and other plants along stream and river margins, in coastal areas, and on a wide range of garden plants.

E. postvittana has a very wide host range, with 73 listed from Australia (Danthanarayana, 1975; Geier and Briese, 1981), and a larger number from New Zealand (Thomas, 1989; Dugdale and Crosby, 1995). Danthanarayana et al. (1995) have suggested that the better performance of *E. postvittana* on herbaceous rather than woody plants suggests that it primarily evolved as a feeder on the former. In Australia, capeweed, curled dock, and plantain are important hosts. In New Zealand, important perennial weed hosts are gorse (*Ulex europeus*) and broom (*Cytisus scoparius*), and in several regions it has been commonly recorded on annual weeds (*Rumex obtusfolius* and *Plantago* spp.) and on shelter and amenity trees (species of *Salix* and *Populus*) (Suckling et al., 1998).

Major Hosts

Acacia spp. (wattles*), Actinidia chinensis* (kiwi), *Chrysanthemum* x *morifolium* (chrysanthemum (florists')), *Citrus* spp., *Cotoneaster* spp., *Crataegus* spp. (hawthorns), *Diospyros* spp. (malabar ebony), *Eucalyptus* spp. (eucalyptus tree), *Feijoa sellowiana* (feijoa fruit), *Humulus lupulus* (hops), *Jasminum* spp. (jasmine), *Ligustrum vulgare* (privet), *Litchi chinensis* (leechee), *Malus pumila* (apple), *Medicago sativa* (alfalfa), *Persea americana* (avocado), *Pinus* spp. (pines), *Pinus radiata* (radiata pine), *Populus* spp. (poplars), *Prunus armeniaca* (apricot), *Prunus persica* (peach), *Pyrus* spp. (pears), *Ribes* spp. (currants), *Rosa* spp. (roses), *Rubus* spp. (blackberry, raspberry), *Solanum tuberosum* (potato), *Trifolium* spp. (clovers), *Vaccinium* spp. (blueberries), *Vicia faba* (broad bean), and *Vitis vinifera* (grapevine) (CABI, 2004).

Known vectors (or associated organisms)

An association between larvae of *E. postvitanna* and *Botrytis cinerea*, grey mold, has been shown in grapes.


Figure 3. Discolored, shriveled berries caused by *Botrytis* bunch rot (left) and *Botrytis cinerea* sporulating on grape berries. Photos courtesy P.Sholberg, Agriculture & AgriFood Canada.

Known Distribution

E. postvittana is widespread throughout Australia and New Zealand on many weedy hosts including gorse (*Ulex europaeus*) and broom (*Cytisus scoparius*). It is commonly present in gardens and unsprayed horticultural crops.

Europe: United Kingdom. **North America:** United States. **Oceania:** Australia, New Caledonia, and New Zealand.

Potential Distribution within the United States

E. postvittana has been reported from Hawaii since the late 1800s. On March 16, 2007, *E. postvittana* was confirmed in Alameda County, California. As of July 2007, further detections have occurred in Alameda, Contra Costa, Los Angeles, Marin, Monterey, Santa Clara, Santa Cruz, San Francisco, San Manteo, and Solano Counties. APHIS and the California Department of Food and Agriculture are conducting delimiting surveys to determine the area of infestation in the State of California. Vennette et al. (2003) estimate that approximately 80% of the continental United States may be climatically suitable for *E. postvittana.*

A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 4) indicates that most states in the United States have a risk rating of 5 or greater for *E. postvittana* establishment based on host availability and climate within the continental United States.

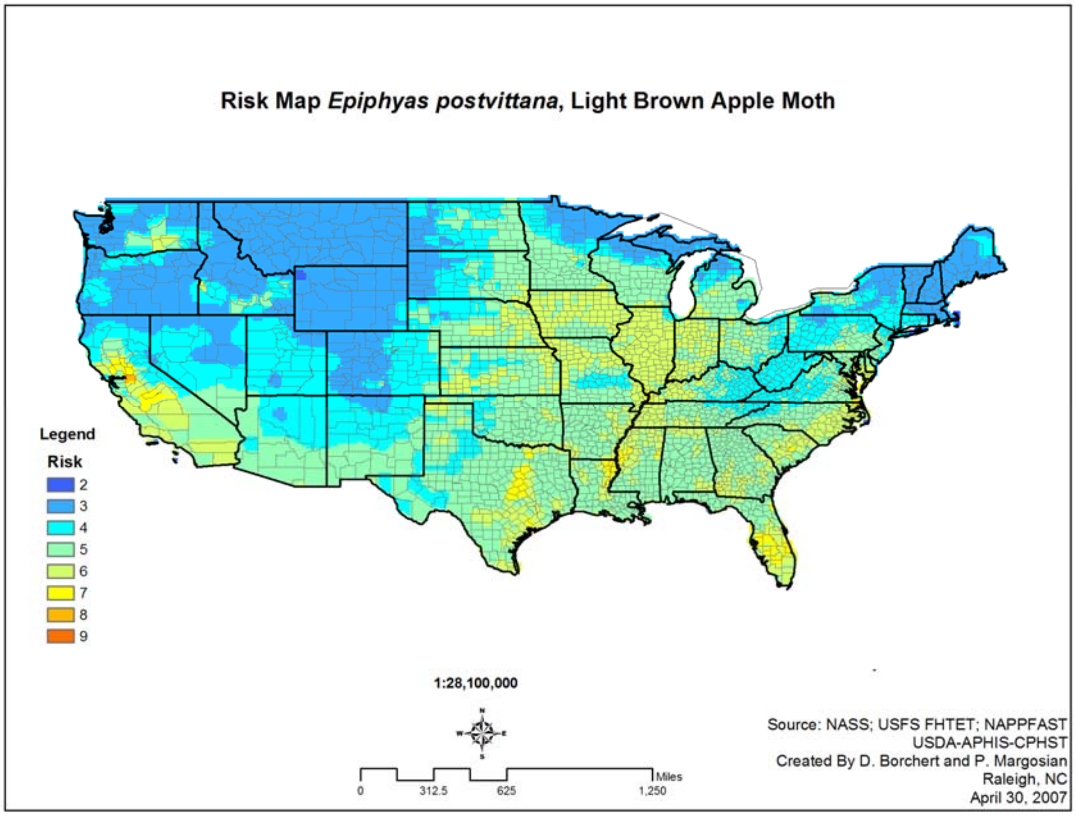

Figure 4. Risk map for *E. postvittana* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Survey

(Taken from Venette et al., 2003 and CABI, 2004)
<u>Preferred Method:</u> Pheromone traps have been widely used for detection and monitoring of populations of this species (Bellas et al., 1983). Two key components of the pheromone are (*E*)-11-tetradecenyl acetate and (*E*,*E*)-(9,11) tetradecadienyl acetate (Bellas et al., 1983). These compounds in a ratio of 20:1 are highly attractive to males. **This lure is available from the CPHST- Otis lab in a 20:1 ratio.** This lure is typically formulated on a rubber septum (1 to 3 mg). Due to the recent detections of *E. postvittana* in California, new formulations (*e.g.,* plastic laminate) are under development and testing is planned at Otis. Delta traps are typically used and placed from 5 to 6.5 ft (1.5 to 2 m) above ground level.

Foster and Muggleston (1993) provide a detailed analysis of different designs of delta traps. In general, they found that traps with a greater length (*i.e.*, the distance between the two openings of the trap) capture significantly more *E. postvittana* than shorter traps. This effect is not related to saturation of smaller sticky surfaces with insects or other debris. The addition of barriers to slow the exit of an insect from a trap also improves catch. In a separate analysis, Foster et al. (1991) found that placing the pheromone lure on the side of the trap helped to improve trap efficiency. The orientation of the trap relative to wind direction did not affect the number of *E. postvittana* that were attracted to the pheromone or were subsequently caught by the trap (Foster et al., 1991).

Alternative Method: Visual inspections have been used to monitor population dynamics of *E. postvittana* eggs and larvae. In grape, 40 vines were inspected per sampling date (Buchanan, 1977). In apple and other tree fruits, 200 shoots and 200 fruit clusters (10 of each on 20 different trees) are often inspected (Bradley et al., 1998). Egg masses are most likely to be found on leaves (USDA ,1984). The egg masses may be jet black if parasitized by *Trichogramma* spp. (a trichogrammatid wasp) (Glen and Hoffman, 1997). Larvae are most likely to be found near the calyx or in the endocarp; larvae may also create “irregular brown areas, round pits, or scars” on the surface of a fruit (USDA, 1984). Larvae may also be found inside furled leaves, and adults may occasionally be found on the lower leaf surface (USDA, 1984).

Not recommended: Adults are also attracted to fruit fermentation products as a 10% wine solution has been used as an attractant and killing agent for adults (Buchanan, 1977; Glenn and Hoffmann, 1997). The dilute wine (670 ml) in 1 liter jars was hung from grapevines on the edge of a block of grapes (Buchanan, 1977). Black light traps have been used to monitor adults of *E. postvittana* (Thwaite, 1976).

Key Diagnostics

E. postvittana is similar to *E. pulla* (Turner) and *E. liadelpha* (Meyr.), both not known to be present in the United States. Geier and Springett (1976) reported possible hybridization based on demographic characteristics. Larvae are similar to larvae of other leafrollers, which may be present (for example, in New Zealand, *Planotortrix octo*, *P. excessana, Ctenopseustis obliquana,* and *C. herana* may be present). Identity of the species must often be confirmed by examination of adult genitalia. Molecular diagnostics based on PCR amplification of ribosomal DNA have been developed and are especially useful for the identification of immature specimens (Armstrong et al., 1997).

Easily Confused Pests

E. postvittana may be confused with *Amorbia emigratella* (Mexican leafroller), which has been reported from the United States, however, *E. postvittana* has ocelli which are lacking in *A. emigratella.* The undersides of *E. postvittana* hindwings are conspicuously immaculate as in *A. emigratella*, and the second abdominal tergite lacks the conspicuous median pit near the base which is present in *A. emigratella* (USDA, 1984).

Figure 4. Larva and adult of *Amorbia emigratella*. Photos courtesy of Laurie Henneman, Department of Biological Sciences, University of Bristol. http://www.umwestern.edu/shares/envirosci_share/laurie/lepidoptera/amorbia.htm

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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: AHP Prioritized Pest List

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

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

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 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).

Biology and Ecology

H. arator is a polyphagous, univoltine pest of pasturelands, turf, and agricultural crops in Australia, New Zealand, and Africa. These scarab beetles spend their entire lifecycle belowground, with the exception of the adult stage (Matthiessen and Learmonth, 1998) (Fig. 1). In spring, the majority of mating occurs, although some may ensue in fall. During this time, adults crawl on the soil surface at night, and flying is limited. Larvae mature in midsummer. Adults emerge after about two weeks in summer to late autumn. The adults are usually found on or under the soil surface, to a depth of about 150 mm. They are a shiny black and cylindrical cockchafer that is slow moving and is approximately 15 mm long. The adult is capable of flying, which serves to disperse the beetle to new sites (Matthiessen and Learmoth, 2005). Wet conditions during the egg and first instar larval stages are fatal, but as the larvae grow, their ability to cope with high moisture levels increases (Matthiessen and Learmoth, 2005).

Symptoms/Signs

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. In woody vines (*e.g.*, grape) and eucalyptus, this type of damage occurs most frequently, causes greater growth distortion, and is potentially fatal to newly planted cuttings or seedlings. African black beetles eat the cuttings and rootlings at or just below ground level, ring barking of the vine, and causing wilting and collapse. The chewing is more likely to be sufficiently deep or to extend more fully around the circumference of the thinner stems at early stages of plant growth. The problem is greatest where vines have been planted onto old pasture land, especially if kikuyu (*Pennisetum clandestinum*) is present. High densities of *H. arator* in pastures lead to clover (a non-host) becoming dominant over grasses (Matthiessen and Learmoth, 2005).

Pest Importance

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).

Impact on newly planted grapevine and eucalyptus seedlings can be severe in patches within a vineyard or plantation, leading to areas of total loss amongst the plant stand. Heavy damage to perennial pasture can be caused by *H. arator* build-up in years with a drier than average spring and early summer, causing greater than usual survival of first-instar larvae (King et al., 1981). These climate-driven outbreaks are characteristic of regions that typically have a wet summer, such as the North Island of New Zealand and eastern Australia. Across the regions infested by African black beetle, this insect can cause significant economic damage to horticultural crops such as young vines (newly planted cuttings and young, rooted vines), olives, and potatoes. **In grape, damage primarily occurs in the first two years after planting because after this time the vines become too woody to be damaged by the beetle. However, older vines may still be damaged, especially if they have been stressed.** The impact of losing young vines is twofold, including replanting costs (especially if grafted vines are involved), and loss of yield through delayed grape production. The unevenness in vine maturity in the block presents management problems, for example, in terms of weed control and vine training. Partial damage to vines by African black beetle can result in retarded growth and add to the cost of vine training because of the prolonged time that such vines require individual attention (CABI, 2004; Matthiessen and Learmoth, 2005;).

Known Hosts

Major hosts

Eucalyptus spp. (Eucalyptus tree), *Lolium perenne* (perennial ryegrass), pastures, *Solanum tuberosum* (potato), *Vitis vinifera* (grape), and *Zea mays* (corn) (CABI, 2004).

Minor hosts

Ananas comosus (pineapple), *Begonia* spp. (begonia), *Brassica napus* (turnip), *Brassica oleracea L.* var. *capitata* (cabbage), *Bromus catharticus* (prairie grass), *Calendula* spp. (marigold), *Cucurbita* spp. (squash), *Daucus carota* (carrot), *Elymus repens* (couch grass), *Eucalyptus saligna* (blue gum), *Fragaria* x *ananassa* (strawberry), *Lactuca sativa* (lettuce), *Lycopersicon esculentum* (tomato), *Olea* spp. (olives), *Paspalum nicorae* (Brunswick grass), *Pennisetum clandestinum* (kikuyu grass), *Petunia* spp. (petunia), *Phaseolus vulgaris* (bean), *Phlox* spp. (phlox), *Pisum sativum* (pea), *Protea* spp. (protea), *Rheum rhabarbarum* (rhubarb), *Secale montanum* (perennial rye), *Sorghum* spp. (sorghum), *Triticum aestivum* (wheat), and *Saccharum officinarum* (sugarcane) (CABI, 2004).

Known Vectors (or associated insects)

H. arator is not a known vector and does not have any associated organisms.

Known Distribution

Africa: Angola, Botswana, Comoros, Congo, Congo Democratic Republic, Ethiopia, Kenya, Lesotho, Madagascar, Malawi, Mozambique, Namibia, Saint Helena, South Africa, Tanzania, Zaire, Zambia, and Zimbabwe; **Oceania:** Australia, New Zealand, Norfolk Island, and Papau New Guinea (CABI, 2004).

Potential Distribution within the United States

The current distribution in Australia and New Zealand of *H. arator* indicates that many regions in the United States may be climatically suitable for the beetle. It is found throughout coastal mainland Australia (north to Brisbane and south to Melbourne) and found in coastal South and Western Australia. *H. arator* is also a pest on the north island of New Zealand. Computer projections for Australia indicate a potential distribution from northern Queensland to southern Tasmania. These areas would correspond to plant hardiness zones 7 through 11 in the United States.

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. African black beetles eat the cutting and rootlings at or just below ground level, ring bark the vine, and cause wilting and collapse. 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: AHP Prioritized Pest List

Pest Description
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 grey and translucent with iridescent glints. The chorion is macroscopically smooth but presents a slight polygonal reticulation in the border and around the micropile. The time elapsed since the eggs were laid may be estimated by observing the eggs: there are five phases of embryonic development - visible embryo, visible eyes, visible mandibles, brown head, and black head. 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. 1A) 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 (CABI, 2004).

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. 1B). Pupal age may be estimated as a function of tegument transparency and coloring (CABI, 2004). 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.

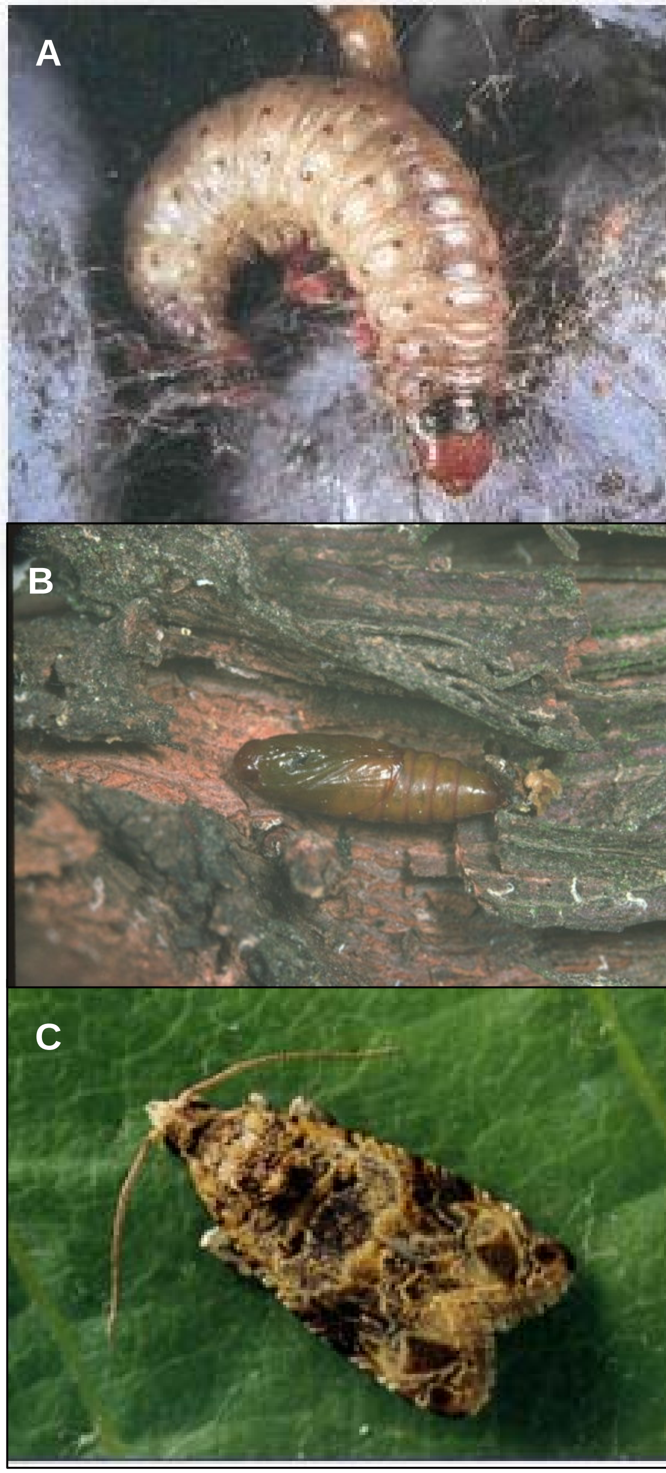

Figure 1. Larva (A), pupa (B), and adult (C) *L. botrana.* Photos courtesy of Instituto Agrario S. Michele All' Adigen, HYPPZ Zoology, and pherobase.net respectively

<u>Adult:</u> Adults are 6 to 8 mm long with a wingspan of about 10 to 13 mm. Adult size is greatly affected by larval food quality (Torres-Vila, 1995). 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. 1C). The ground color is bluish grey 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 grey, gradually darker towards the costa and apex. Hindwings are light brownish grey, darker towards the apex. Cilia and cubital tuft are greyish brown with a paler basal line. The underside is a uniform light grey. There is no clear sexual

dimorphism, but the sexes may be easily separated by their general morphology and behavior. In the pupal stage, males are smaller than females; they have a narrower abdomen with a fine anal comb of modified scales (hair pencils); and when disturbed, they move more quickly and nervously than females (CABI, 2004).

Figure 2. Adult on grape fruit (A) and larvae feeding inside a grape (B). Photos courtesy of Michael Breuer. http://www.bio-pro.de/de/region/freiburg/magazin/01476/index.html

Biology and Ecology:
The first flight of adults occurs in spring when daily average air temperature is above the minimal threshold temperature of 10 °C for 10 to 13 days. The second flight period begins in summer (USDA, 1985). In Israel, adults appear in the vineyard when grapevines flower. Adults are hard to discover during the day and may be noticed only when they take flight after being disturbed. They fly at dusk whenever the temperature is above 12 °C, but rainfall and wind will reduce flight. Adults usually prefer hot, dry places protected from wind so they fly mainly between the first rows of grapevines close to windbreaks and on slopes facing the sun (Avidov and Harper, 1969).

Within a day or two of mating, females begin to oviposit on the blossoms, leaves, and tender twigs of the grapevine. The female lays 300 or more eggs singly at a rate of more than 35 per day. During rearing experiments under laboratory conditions in Czechoslovakia, the optimum temperatures for oviposition were from 20 to 27 °C (Gabel, 1981). First generation eggs are laid on the flower buds or pedicels of the vine while second generation eggs are laid on individual grapes (USDA, 1985) (Fig. 2A). Eggs hatch in 7 to 11 days in spring and 3 to 5 days in summer.

The European grape berry moth is a polyvoltine species (CABI, 2004). The number of generations in a given area is fixed by photoperiod together with temperature, acting on diapause induction and development rate, respectively. Short-day photophases (between 8 and 12 h) during the larval stage induce diapause in larvae that will be later expressed in pupae. The moth achieves two generations in northern cold areas and usually three generations in southern temperate areas, although this general latitudinal pattern is often modified by the altitude-derived gradient and/or microclimatic conditions in a given area. Thus the number of generations has a broader range, reported as one generation in Romania (Filip, 1986) to four generations (often partial) in Spain, Greece, Crete, Italy, and former Yugoslavia (Coscolla, 1997 and references therein). Five generations have been reported in Turkmenistan (Rodionov, 1945).

Figure 3. Glomerules of *L. botrana.* Photo courtesy of EFAPO-ES. http://www.efa-dip.com

First generation larvae feed on bud clusters or flowers and spin webbing around them (glomerules) before pupating inside the web or under the rolled leaf. Second generation larvae enter the grapes (Fig. 2B) and feed before pupating inside the grape. Larvae of the third generation, the most damaging, feed on ripening grapes, migrating from one to another and spinning webs. The third generation larvae leave the fruit and shelter under the bark, among dead leaves, or between clods of earth, where they pupate before overwintering. Few of these larvae pupate before harvest, and many are gathered with the grapes. Larvae develop in 4 to 5 weeks in spring and 2 to 3 weeks in summer. Pupation lasts 9 to 12 weeks in spring 5 to 7 days in summer, and up to 6 months in winter (CABI, 2004).

Moth activity (*i.e.*, flight, feeding, calling, mating, and egg-laying) is principally displayed at dusk, although some activity can also occur at daybreak or at any time on cloudy days. Water availability is necessary for adults to reach their potential reproductive output (Torres-Vila et al., 1996). Females are usually monandrous, but several physiological factors may enhance multiple mating (Torres-Vila et al., 1997). On the other hand, males are largely polygynic (Torres-Vila et al., 1995).

Symptoms/Signs

On grape 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. 3), 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. On grapes (summer generations), larvae feed externally and when berries are a little desiccated (Fig. 4), they penetrate them, bore into the pulp, and remain protected by the berry peel (Fig. 2B, 5). Larvae secure the pierced berries to surrounding ones by silk threads in order to avoid falling. Each larva directly damages several berries (one to six), but if the conditions are suitable for fungal or acid rot development, a large number of berries placed around may be also affected. **Damage is variety-dependent; generally it is more severe on grapevine varieties with dense grapes because this increases both larval installation and rot development.**

On both inflorescences and grapes, several larvae may co-exist in a single reproductive organ. Larval damage on growing points, shoots, or leaves is unusual.

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 generation results 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).

Figure 4. Damage by *L. botrana*. Photo courtesy of HYPPZ Zoology.

Pest Importance

The European grape-berry moth is a serious pest in the warm vine-growing countries where it is normally found. Larvae feed on flower buds, developing berries, and most destructively, on the ripening fruit of grape. The primary damage to grape berries attracts other insects and predisposes the fruit to fungal infection. Larval boring in grapes may promote a number of fungal rots, including *Aspergillus*, *Alternaria*, *Rhizopus*, *Cladosporium*, *Penicillium* and especially, the grey rot caused by *Botrytis cinerea* (CABI, 2004). Loss of up to one-third of the vintage has been reported in areas of the Soviet Union, Syria, and Yugoslavia. Losses in Israel sometimes reach 40 to 50 percent among table grapes and up to 80 percent or more for wine grapes. Further loss is due to the time and labor spent in cleaning the grape bunches. When infestations are heavy, the work days spent in cleaning the fruit account for 30 to 40 percent of the time of those involved in harvesting (USDA, 1985).

Figure 5. Larva inside grape fruit. Photo courtesy P. del Estal (CABI, 2004).

On grapes (summer generations), indirect damage is usually more important than direct, at least in the event of less severe attacks. Thus global damage may appear of little importance if it is evaluated exclusively as weight loss (direct damage) because greater damage is due to rot-derived reduction in quality (indirect damage). Larval boring in grapes may promote a number of fungal rots including *Aspergillus*, *Alternaria*, *Rhizopus*, *Cladosporium*, *Penicillium,* and especially *Botrytis cinerea* (Fig. 6) (Fermaud and Le Menn, 1989; CABI, 2004).


Figure 6. Discolored, shriveled berries caused by Botrytis Bunch Rot (left) and *Botrytis cinerea* sporulating on grape berries (right). Photos courtesy P.Sholberg, Agriculture & AgriFood Canada.

Known Hosts

This pest feeds primarily on the flowers and fruits of grapes. However, *L. botrana* demonstrates a curious behavior of feeding on many different plant families (approximately 27), but only a few species within each family are suitable. Grape cultivars with prolonged blossoming or late-ripening berries are usually more heavily infested than short-flowering or early ripening varieties (Avidov and Harper, 1979). *L. botrana* exhibits an oviposition preference for privet and certain grape cultivars, such as 'Cabernet Sauvignon' (Maher et al., 2000, 2001).

Major hosts
Vitis vinifera (grape), *Vitis* spp.

Minor hosts
Actinidia chinensis (kiwi), *Clematis vitalba* (traveler's joy), *Coffea* spp. (coffee), *Dianthus* spp. (carnation), *Diospyros kaki* (persimmon), *Hordeum vulgare* (barley), *Medicago sativa* (alfalfa), *Olea europaea* subsp. *europaea* (olive), *Prunus amygdalus*, *Prunus avium* (sweet cherry), *Prunus domestica* (plum), *Prunus spinosa* (blackthorn), *Punica granatum* (pomegranate), *Pyrus communis* (pear), *Ribes nigrum* (blackcurrant), *Ribes rubrum* (red currant), *Ribes uva-crispa* (gooseberry), *Rosa* spp. (rose), *Rubes fruticosus* (European blackbetty), and *Solanum tuberosum* (potato).

Wild hosts
Arbutus unedo (arbutus), *Berberis vulgaris* (European barberry), *Clematis vitalba* (old

man's beard), *Cornus mas* (cornelian cherry), *Cornus sanguinea* (dogwood), *Daphne gnidium*, *Galium mollugo* (smooth bedstraw), *Hedera helix* (ivy), *Lamium amplexicaule* (henbit), *Ligustrum vulgare* (privet), *Lonicera tatarica* (Tatarian honeysuckle), *Menispermum canadense* (common moonseed), *Parthenocissus quinquefolia* (Virginia creeper), *Rhus glabra* (smooth sumac), *Rosmarinus officinalis* (rosemary), *Rubus caesius* (dewberry), *Rubus fruticosus* (blackberry), *Syringa vulgaris* (lilac), *Tanacetum vulgare* (tansy), *Trifolium pretense* (red clover), *Viburnum lantana* (wayfaring tree), and *Ziziphus jujuba* (common jujube)

Known Vectors (or associated organisms)

L. botrana has also been shown that the nutritional alteration of berries caused by *Botrytis cinerea* may enhance female fecundity (Savopoulou-Soultani and Tzanakakis, 1988). Larval boring in grapes may promote a number of fungal rots including *Aspergillus*, *Alternaria*, *Rhizopus*, *Cladosporium*, *Penicillium,* and especially *Botrytis cinerea.*

Known Distribution

In 2008, first report of *L. botrana* in Western Hemisphere (South America, Chili).

Africa: Algeria, Egypt, Eritrea, Kenya, Libya, and Morocco; **Asia:** Armenia, Azerbaijan, Georgia, Iran, Israel, Japan, Jordan, Kazakhstan, Lebanon, Syria, Tajikistan, Turkey, Turkmenistan, and Uzbekistan; **Europe:** Austria, Bulgaria, Cyprus, Czech Republic, Czechoslovakia, France, Germany, Greece, Hungary, Italy, Luxembourg, Macedonia, Malta, Moldova, Portugal, Romania, Russia, Serbia and Montenegro, Slovakia, Slovenia, Switzerland, Ukraine, and the United Kingdom (CABI, 2004). **South America**: Chili.

Potential Distribution within the United States

Climatic conditions in the major grape growing areas of the United States favor the establishment of *L. botrana* (USDA, 1985; Venette et al., 2003). Venette et al. (2003) estimate that approximately 29% of the continental United States may be suitable for *L. botrana*. This projection includes the major California wine-producing counties of Napa, Sonoma, Amador, Monterey, and San Luis Obispo.

Survey

From Venette et al. (2003)

<u>Preferred Method:</u> 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 CPHST- Otis lab. 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; with a sticky area of 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).

<u>Alternative Method:</u> 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 (Fig. 8) 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.

<u>Not recommended:</u> 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*. Even the primarily phytophagous *Sparganothis pilleriana* may sometimes damage grapes. However, only the first of these, *E. ambiguella* (Fig. 7), 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 greyish white cocoon that usually does not incorporate vegetal residues and frass, as occurs in *E. ambiguella* (CABI, 2004).

Figure 7. *Eupoecilia ambiguella* adult and pupa. Photos courtesy of HYPP Zoology.

Figure 8. American grape berry moth, *Endopiza viteana*. Photo courtesy of Michigan State University.

Another tortricid species, the American grape berry moth, *Endopiza viteana* (Fig. 8) [*Polychrosis viteana*], occurs in the eastern USA and presents similar bionomics to *L. botrana* (Venette et al., 2003).

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Planococcus minor

Scientific Name
Planococcus minor Maskell

Synonyms:
Planococcus pacificus, Pseudococcus minor, Dactylopius calceolariae minor, Planococcus psidii, and *Pseudococcus calceolariae minor.*

Common Name(s)
Passionvine mealybug, Pacific mealybug

Type of Pest
Mealybug

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Pseudococcidae

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

Pest Description
Planococcus minor is a small sucking insect with a cottony appearance. Females are oval, 1.3 to 3.2 mm long. The insect body is distinctly segmented, yellow to pink in color, and covered with powdery wax, with the appearance of "having been rolled in flour" (Fig. 1A). The margin of the body has a complete series of 18 pairs of cerarii, each cerarius with 2 conical setae (except for preocular cerarii which may have 1 or 3 setae). Legs are elongate.

It is assumed that this species is identical in appearance to *P. citri* (Fig. 1B) as follows: body oval; slightly rounded in lateral view; body yellow when newly molted, pink or orange-brown when fully mature; legs brown-red; mealy wax covering body, not thick enough to hide body color; with dorsomedial bare area on dorsum forming central longitudinal stripe (more obvious than on *P. ficus*); ovisac ventral only, may be 2 times longer than body when fully formed; with 17 or 18 lateral wax filaments, most relatively short, often slightly curved, posterior pair slightly longer, filaments anterior of posterior pair small, posterior pair about 1/8 length of body. Primarily occurring on foliage of host. Oviparous, eggs yellow. Surface of lateral filaments rough (Rung et al., 2007).

Biology and Ecology:
With the exception of a few species such as *Planococcus citri*, details about life stages of many mealybugs, particularly *P. minor*, are not well known. *Planococcus citri* has 4-8 generations annually, and within *Planococcus* species, there are typically 4 instars for females and 5 for males (McKenzie, 1967; Williams, 1985). In Israel, generation time ranges from 4-6 weeks during summer months and approximately 3 months in winter (Mendel and Blumberg, 2004). Development rate and the number of generations is highly variable, and is determined by several factors including plant host selection and feeding site as these relate to nutrition, temperature, population density of the mealybug complex, and the presence of predators (McKenzie, 1967; Miller and Kosztarab, 1979; Williams, 1985). Population density may also vary depending on the presence of ants (several genera) that are known to have an association with *Planococcus* species. Ants have been observed feeding on the honeydew excretions of mealybugs and protecting this important food source from predators. Ants may also play a role in mealybug dispersal. Mealybug populations closely associated with ants tend to be larger than non-tended populations of the same species (McKenzie, 1967; Lamb, 1974; Youdeowei and Service, 1983; Buckley and Gullan, 1991).

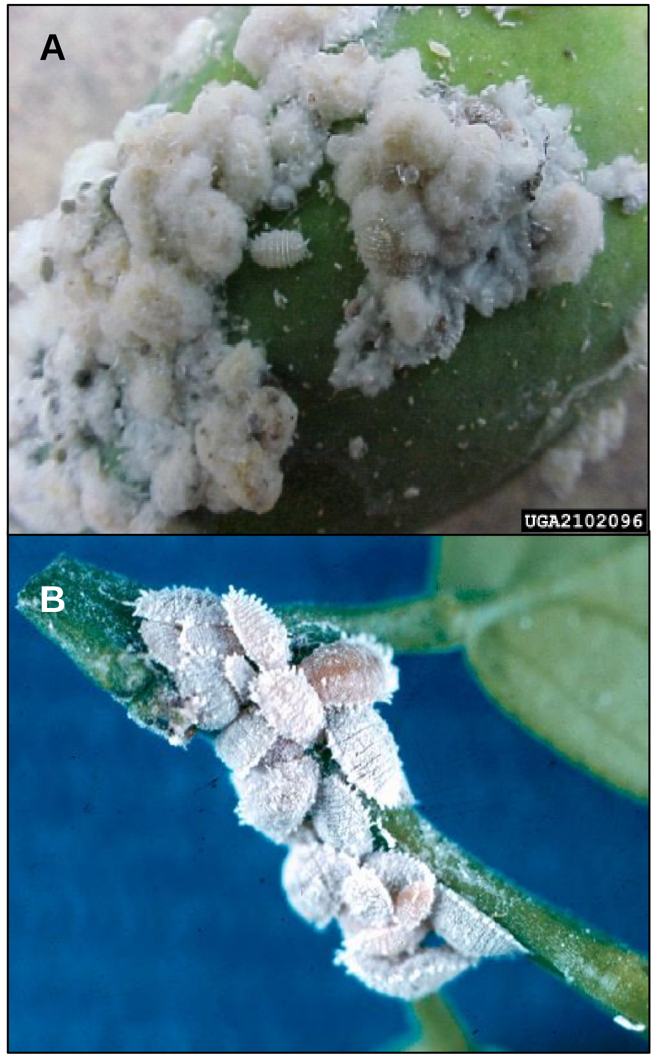

Figure 1: *Planococcus minor* (A) and *P. citri* (B). Photos courtesy of Jeel Miles (www.invasive.org) and J. V. French.

Most species are thought to reproduce sexually, though some parthenogenesis may occur. To further add to the complexity, hybrid crosses of *P. citri* and *P. ficus* have been observed under laboratory conditions, which suggests that similar hybridization may occur among closely related species in a mealybug complex under natural conditions (Williams, 1985).

In laboratory studies, *P. minor* females produced between 65-425 eggs on varying hosts (Maity et al., 1998; Martinez and Suris, 1998; Biswas and Ghosh, 2000). Preoviposition period ranged from 8-12 days, and the incubation period lasted approximately 3 days. The time to complete 1 generation ranged from 31-50 days. The median development time for males was slightly longer in duration than for females.

Pest Importance

The passionvine mealybug is a foreign plant pest that attacks over 240 different species of plants, including both agricultural and ornamental plants. It has invaded areas in American Samoa, the U. S. Virgin Islands, and Mexico. This pest could enter the eastern United States from Mexico or from the Caribbean or enter California from Mexico or from the Pacific (Vennette and Davis, 2004). The dispersal potential considers both the number of offspring and motility of the pest. On mandarin, this insect completed 10 generations per year and averaged 260 eggs per generation (Sahoo et al., 1999). Local distribution was limited, but over 1,900 interceptions of this pest on various hosts from over 30 countries were reported from 1985 to 2000.

The two polyphagous mealybugs, *Planococcus minor* and *P. citri*, have similar host ranges and distributions within the Neotropical region and may simultaneously infect the same plant. The predominant species in the South Pacific Islands, the Austro-oriental Region, the Malagasian region, and the Northern Neotropical Region is *P. mino*r, as opposed to *P. citri*, which is present southern United States and reported as far north as Ohio, Kansas, and Massachusettes (Cox, 1989). In addition to direct damage, *P. citri* was reported as a virus vector in cocoa, banana, and grape, but whether *P. minor* can serve as a vector is unknown (Jones and Lockhart, 1993; Canaleiro and Segura, 1997).

Symptoms/Signs

Mealybugs have piercing-sucking mouthparts. *Planococcus minor* is a phloem feeder, and in general this may cause reduced yield, reduced plant or fruit quality, stunting, wilting, discoloration, and defoliation. Indirect or secondary damage is caused by sooty mold growth on honeydew produced by the mealybug.

Known Hosts

Planococcus minor has a broad host range and is considered a non-discriminate feeder within a number of plant families. More than 250 host plants have been identified. Economically important hosts are identified below. For a complete listing of hosts see Venette and Davis (2004) and Scale Net (http://www.sel.barc.usda.gov/scalenet/scalenet.htm).

Major hosts
Citrus spp. (grapefruit, orange, lemon, lime, sour orange), *Coffea* spp. (coffee), *Colocasia esculenta* (taro), *Mangifera indica* (mango), *Musa* spp. (banana), *Psidium guajava* (guava), *Theobroma cacao* (cacao bean), *Vitis* spp. (grape), and *Ziziphys* spp. (jujube).

Minor hosts
Anacardium occidentale (cashew), *Ananas comosus* (pineapple), *Apium graveolens* (celery), *Arachis hypogea* (peanut), *Brassica oleracea* (cabbage), *Cajanus cajan*

(pigeon pea), *Capsicum* spp. (pepper), *Cucumis melo* (melon), *Curcurbita* spp. (pumpkin, squash), *Ficus* spp. (fig), *Fragaria* spp. (strawberry), *Glycine max* (soybean), *Ipomoea batatus* (sweet potato), *Lycopersicon esculentum* (tomato), *Morus* spp. (mulberry), *Ocimum* spp. (basil), *Oryza sativa* (rice), *Persea americana* (avocado), *Phaseolus* spp. (bean), *Saccharum officinarum* (sugarcane), *Solanum melongena* (eggplant), *Solanum tuberosum* (potato), *Vigna* spp. (cowpea), and *Zea mays* (corn).

Known Vectors (or associated organisms)

Mealybugs produce honeydew, which is a liquid rich in sugar. Ants like to feed on honeydew and some ants will, therefore, protect the mealybugs by chasing away predators and parasitoids. The ants also carry mealybugs around and thus contribute to their distribution. *P. citri* was reported as a virus vector in cocoa, banana, and grape, but whether *P. minor* can serve as a vector is unknown (Jones and Lockhart, 1993; Canaleiro and Segura, 1997).

Known Distribution

Africa: Guinea, Madagascar, Rodrigues Island (Mauritius), and Seychelles. **Asia:** Andaman Islands, Bangladesh, Borneo, Brunei, Burma, Cambodia, India, Indonesia, Malaysia, Maldives, Philippines, Singapore, Sri Lanka, Taiwan, Thailand, and Vietnam. **North America:** Mexico. **Central America:** Costa Rica, Guatemala, and Honduras. **South America:** Argentina, Brazil, Columbia, Galapagos Islands, Guyana, Suriname, Trinidad and Tobago, and Uruguay. **Caribbean:** Bermuda, Cuba, Dominican Republic, Grenada, Guadeloupe, Haiti, Jamaica, Saint Lucia, and US Virgin Islands. **Oceania:** American Samoa**,** Australia, Cook Islands, Fiji, French Polynesia, Guam, New Caledonia, Niue, Papua New Guinea, Pohnpei, Samoa, Solomon Islands, Toklelau, Tonga, and Vanuatu.

Potential Distribution within the United States

***P. minor* is <u>not</u> currently present in the continental United States (see http://www.pestalert.org/viewNewsAlert.cfm?naid=20).** During a routine inspection of tropical plants in a California greenhouse in April 2006, a suspect sample was discovered and initially misidentified as *P. minor* and an alert was posted to the Phytosanitary Alert System of the North American Plant Protection Organization (NAPPO). The initial sample and others from the surrounding area were identified as *P. citri* by the USDA Systematic Entomology Laboratory.

The host range of *P. minor* includes a wide range of plants grown in the United States, so this insect appears capable of establishing populations that mirror the distribution of *P. citri*. *P. citri* is present in the southern states and has been reported as far north as Ohio, Kansas, and Massachusettes. Venette and Davis (2004) estimate that approximately 52% of the continental United States would have a suitable climate for *P. minor.* A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 2) indicates that portions of Arkansas, Texas, Louisiana, Mississippi, Alabama, George, Florida, North Carolina, and South Carolina have the greatest risk for *P. minor* establishment based on host availability and climate within the continental United States.

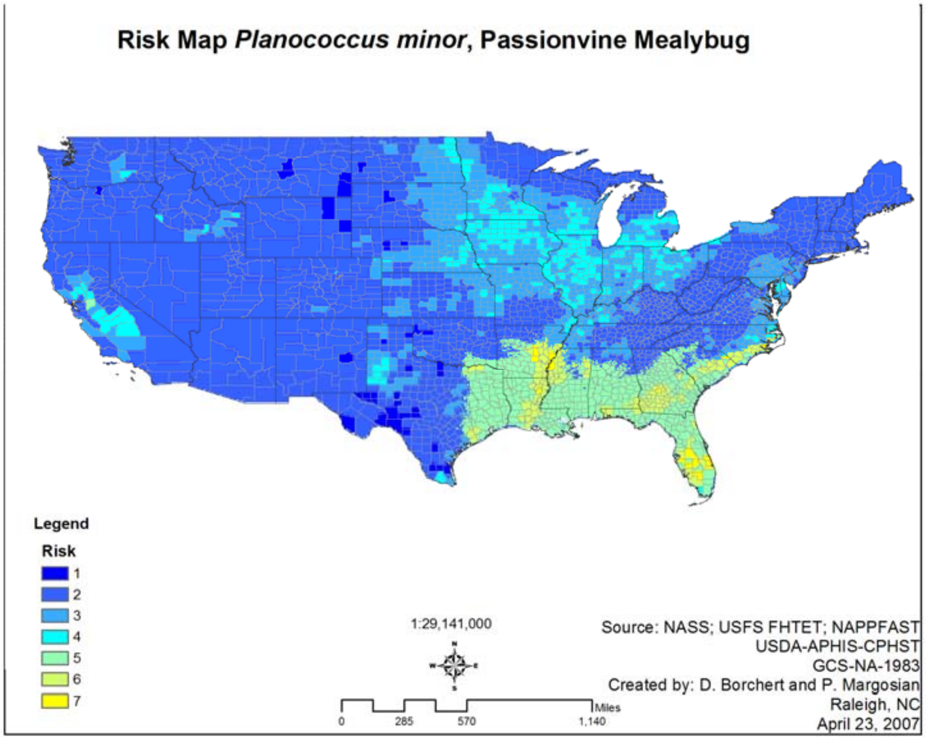

Figure 2. Risk map for *P. minor* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Survey (from Vennette and Davis, 2004)

Preferred Method: In the United States, surveys for mealybugs other than *P. minor* require “time-consuming and often laborious examination of plant material for the presence of live mealybugs” (Millar et al., 2002). No simple, alternative techniques are available (Millar et al., 2002). The same holds true for *P. minor* surveys in other parts of the world. In India, a regional survey for scales and mealybugs, including *P. minor*, was based on visually examining 25 branches or leaves on each of 15 plants collected from each of 3 field sites in 162 locations (25 x 15 x 3 x 162 = 182,250 leaves examined).

Researchers also depend on visual inspections to assess densities of *P. minor*. In a study of *P. minor* population dynamics, populations of the mealybug were evaluated by visual inspection of citrus leaves, specifically 10 to 15 leaves from 10 randomly selected plants (Bhuiya et al., 2000). Reddy et al. (1997) followed a similar protocol for coffee.

No pheromones have yet been identified for *P. minor*. However, previous research on closely related mealybug species suggests that the identification of a sex pheromone and subsequent development of a pheromone-baited trap is highly feasible (Bier-Leonhardt et al., 1981; Millar et al., 2002).

Key Diagnostics (from Vennette and Davis, 2004)

Infestations reduce the vigor and growth of foliage plants, which reduces the beauty of the plant and affects marketability (Hamlen, 1975). Mealybugs are a quarantine problem on exported foliage and flowers. This is due to the fact that species cannot be accurately identified outside of the lab, so inspectors/surveyors should treat all specimens as unknown species. There are a large number of endemic species of mealybugs in the United States and identifications need to be made by a recognized taxonomic authority.

Planococcus species are not easily distinguishable from one another, especially when immature. A level of complexity is added with variable morphological characters in some species; distinguishing morphological characters can change depending on environmental conditions such as temperature. Distinguishable morphological features of closely related mealybug species are described by Cox (1981, 1983, and 1989). A Lucid tool for scale insects has been recently developed, which contains a tool on mealybugs (see http://www.sel.barc.usda.gov/ScaleKeys/ScaleInsectsHome/ScaleInsectsMealybugs.html).

PPQ initiated a project to develop molecular diagnostics to separate *P. citri* from *P. minor*. Considerable progress has been made in developing a PCR-RFLP technique. A final report of this work was completed in June 2007. For further information or to obtain the report, contact Terrence Walters at (terrence.w.walters@aphis.usda.gov) or go to http://ppqwrrpt.aphis.usda.gov:8080/DocMgt/Default.aspx (search author = Rung or title = *Planococcus*).

Easily Confused Pests

Planococcus citri and *P. minor* have been taxonomically confused and routinely misidentified as adults are similar in appearance and share similar hosts and geographic range (Williams, 1985; Cox, 1989; Williams and Granara de Willink, 1992). Adults (females) can be identified based upon close examination of morphological characters by a taxonomist.

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Scirtothrips dorsalis

Scientific Name
Scirtothrips dorsalis Hood

Synonyms:
Anaphothrips andreae, Heliothrips minutissimus, Neophysopus fragariae, Scirtothrips andreae, S. fragariae, S. minutissimus, and *S. padmae*

Common Name(s)
Castor thrips, chilli thrips, yellow tea thrips, grapevine berry thrips, strawberry thrips

Type of Pest
Thrips

Taxonomic Position
Class: Insecta, **Order:** Thysanoptera, **Family:** Thripidae

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

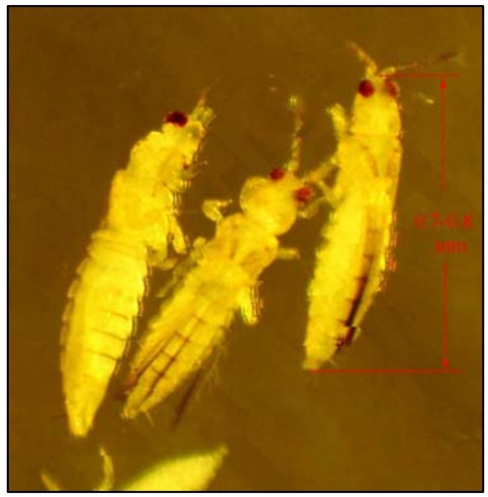

Figure 1. *Scirtothrips dorsalis* adults. Courtesy of T. Skarlinsky (USDA APHIS PPQ)

Pest Description
Scirtothrips dorsalis is a widespread pest, described as a new species by Hood in 1919. *S. dorsalis* is a very small (0.5 to 1.2 mm), pale yellow-colored thrips (Fig. 1) that can be found feeding on leaves, flowers, and calyxes of fruit on a wide variety of host crops. It is difficult to recognize this thrips with the naked eye, and definitive identification is best accomplished at approximately 40 to 80 x magnification.

Eggs: Typically oval, whitish to yellowish, narrow anteriorly, with an incubation period of 6 to 8 days (CABI, 2004). Eggs are about 0.075 mm long and 0.070 mm wide, and are inserted inside plant tissue.

Larvae: Two larval stages (first and second instar) last for 6 to 7 days. The larvae are off-white in color.

> *First instar:* transparent; body short, legs longer; antennae short, swollen; mouth cone bent and short; and antennae seven-segmented and cylindrical. Sclerotization not distinct, head and thorax reticulate (CABI, 2004).
>
> *Second instar*: antennae longer, cylindrical, seven-segmented; mouth cone longer; maxillary palpi three-segmented; body setae longer than the first instar; head and thorax reticulate with sclerotization of head (CABI, 2004).

Pre-pupae: Yellowish; antennae swollen, short, with distinct segmentation; two pairs of external wing buds on each meso- and metathorax (CABI, 2004). The pre-pupal period is short (~24 hours).

Pupae: Dark yellow with eyes and ocelli bearing red pigmentation; wing buds are elongate; antennae short and reflected over the head; female pupae with larger pointed abdomen; males have a smaller, blunt abdomen (CABI, 2004). The pupal period lasts 2 to 3 days. **Pupation takes place in the axils of leaves, in leaf curls, and under the calyxes of flower and fruits.**

Adults: Almost white on emergence, turning yellowish subsequently (Fig. 1) with incomplete dark stripes on the dorsal surface where the adjacent abdominal segments meet. A technical description follows: abdominal tergites with median dark patch, tergites and sternites with dark antecostal ridge; ocellar setae pair III situated between posterior ocelli; 2 pairs of median post-ocular setae present; pronotum with four pairs of posteromarginal setae, major setae 25 to 30 µm long; metanotum medially with elongate recticles or striations, arcuate in anterior third, median setae not at anterior margin; forewings with fore marginal setae, second vein with two setae, cilia straight; tergal microtrichial fields with 3 discal setae, VIII and IX with microtrichia medially; sternites with numerous microtrichia, more than 2 complete rows medially; male without drepanae on tergite IX (Palmer and Mound, 1983).

Biology and Ecology:
Reproduction is both sexual and parthenogenetic. In India, where the life cycle has been studied particularly, adults typically mate 2 to 3 days after their pupal molt and females start ovipositing on *Ricinus* spp. 3 to 5 days after emergence. The total number of eggs laid ranges from 40 to 68 (Venette and Davis, 2004). The life cycle is completed in 15 to 20 days, and the sex ratio is 6:1 females to males. On chillies, a single female lays 2 to 4 eggs per day for a period of about 32 days. In the Guntur area of India, *S. dorsalis* appears in two distinct periods: in the nurseries in August-September, when it is not serious, and from the third week of November to March. *S. dorsalis* typically undergoes 4 to 8 generations per year (Venette and Davis, 2004). A degree day model was developed by Meisner et al. (2005) to determine the potential number of generations per year that the United States climate could support, as well as pinpoint the areas that could climatically support establishment. The model was based on a degree day (DD) requirement of 281 days per generation. In addition the model excluded regions where the minimum temperature reached -4 °C or below for five consecutive days. The results showed that parts of Florida, Texas, Arizona, California, and Nevada could sustain up to 18 generations per year and that several additional states (Louisianna, Alabama, Louisiana, Georgia, and South Carolina) are susceptible to establishment (Fig. 2).

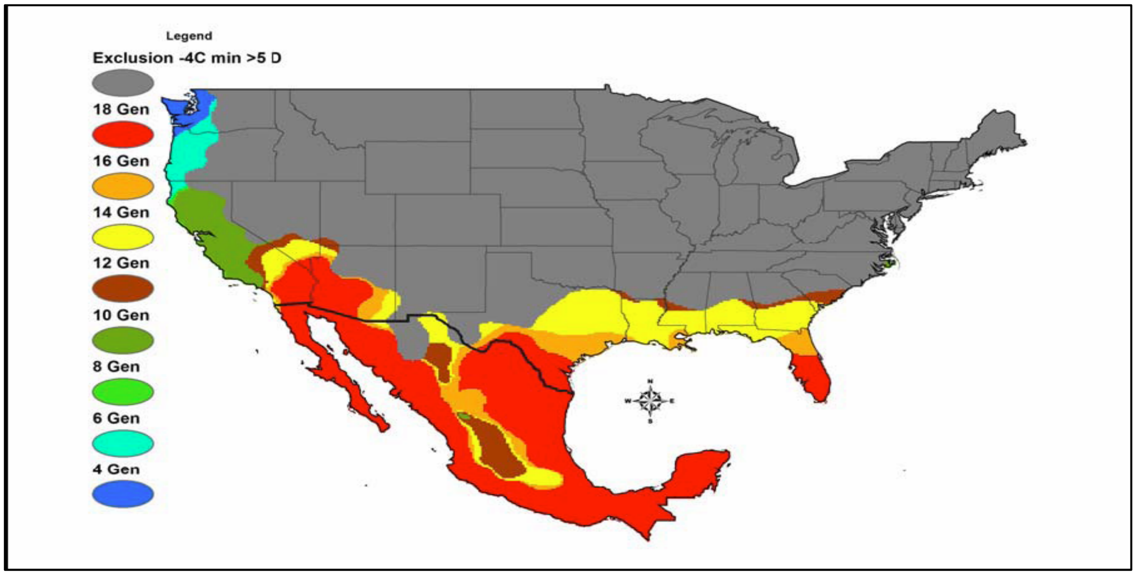

Figure 2. Generational potential (based on a generational requirement of 281 DD and a base and upper development temperature of 9.7°C and 33.0°C, respectively) outside of the predicted cold temperature exclusion boundary (areas where the minimum daily temperature reaches -4ºC or below on 5 or more days per year) for *S. dorsalis* in the United Statesand Mexico (from Meisner et al., 2005).

Pest Importance

S. dorsalis is a significant pest of chilli pepper, citrus, castor bean, cotton, onion, and other crops in tropical and subtropical regions of Asia, Africa, eastern Europe, Oceania, and Japan. Recently, *S. dorsalis* was confirmed for the first time from several Caribbean Islands, Puerto Rico, Florida, and Texas. *S. dorsalis* is widespread in Florida, but it is not known if it is present or established in any other part of the continental United States.

On grapes, heavy feeding damage to flower clusters and developing fruit has resulted in reduced fruit set and reduced marketability of damaged fruits (Ananthakrishnan, 1971). In Asia, *S. dorsalis* is a pest of economic important in pest of citrus and feeding by the thrips causes significant leaf and flower deformation, fruit damage, and yield reduction. In Taiwan, a range of damage on mango, from fruit scarring to total plant defoliation has been reported (Lee and Wen, 1982).*S. dorsalis* is also an economically important pest of chilli pepper, castor bean, cotton, and onion; in these crops, thrips feeding can wilt, distort, or stunt young leaves/shoots and cause premature leaf, bud, or flower drop. In some varieties of chilli peppers, ~75% of leaves may be deformed due to the activity of piercing-sucking insects. Yield losses attributable to *S. dorsalis* in chilli have ranged from 20% (Ahamad et al., 1987) to nearly 50% (Sanap and Nawale, 1987; Varadharajan and Veeravel, 1996). In cotton, sucking pests, including *S. dorsalis*, reduced yield of seed by 77%; fiber yields and quality were also diminished (Gupta and Gupta, 1999). Estimated yield loss has been recorded at 25 to 67% when population density is high. On tea, feeding occurs on new growth, including leaves, shoots and buds, and occasionally on older leaves, resulting in browning and defoliation (Ananthakrishnan, 1971). *Scirtothrips dorsalis* is also considered a major pest of roses in India, where its feeding distorts or destroys leaves, buds, and flowers and reduces marketability (Gahukar, 1999). On mango, feeding damage occurs on new growth, on the underside of leaf surfaces at the midrib, and on fruits (Zaman and Maiti, 1994). Damaged tissues appear dark in color, and leaves curl and drop prematurely (Kumar et al., 1994).

This insect is also a key vector of *Tomato spotted wilt virus* (TSWV), which causes bud necrosis disease (BND), an important disease of peanut in India (Amin et al., 1981; Mound and Palmer, 1981; Ananthakrishnan, 1993; Lewis, 1997). TSWV is widely distributed in the eastern United States on a variety of ornamental, field crops (peanut, tobacco, tomato), and weeds. *S. dorsalis* is also known to vector peanut chlorotic fan-spot virus, peanut yellow spot virus and tobacco streak virus (Rao et al., 2003). It is also reported to transmit the bacterial leaf spot bacterium (Ananthadrishnan, 1993; Mound, 1996; and Mound and Palmer, 1981).

Symptoms/Signs

As with other plant-feeding thrips, damage is caused by sucking out sap from individual epidermal cells, leading to necrosis of the tissue. Damage is most severe at the growing tips, on young leaves and shoots, or on flowers and young fruits. Heavy feeding damage turns tender leaves, buds, and fruits bronze to black in color. Damaged leaves curl upward (Fig. 3) and appear distorted. Infested plants become stunted or dwarfed, and leaves with petioles detach from the stem, causing defoliation in some plants. Symptoms include: silvering of the leaf surface; frass-marked staining or scarring of the fruit, particularly around the apex or at the rim of the calyx; distortion and staining of both leaves (curling and thickening of the lamina) and fruit; and ultimately, the early senescence of the leaves. *S. dorsalis* rarely feeds on mature leaves.

Figure 3. Leaf curling symptom. Photo courtesy of CABI, 2004.

In tropical regions, the abundance of chilli thrips is low in the rainy season, but becomes high during the dry season. As is typical of species in the genus *Scirtothrips*, eggs are laid in the youngest tissues of plants, and feeding by adults and larvae can result in extensive cell damage to these developing tissues, leading to leaf and fruit distortion, and flower fall.

On chillies, *S. dorsalis* causes 'leaf curl disease.' Heavy infestation of the tender shoots, buds and flowers causes the leaves to curl badly (Fig. 3) and drop; and fresh buds to become brittle and subsequently drop down.

On grape, the fruits and leaves are attacked, which causes damage and/or scarring on the leaves (Fig. 4A,) on the surface of the berries (Fig. 4B), on the rachis (Fig. 4C). Berries are attacked, starting at even the button stage, and significant scarring develops if control measures are not taken during berry formation. In severe cases of infestation, the berry bursts and exposes the seeds. The infestation can last until fruit maturation and facilitate the secondary infestation of the fruit by certain flies and fungi. The affected fruit become unfit for consumption, canning, or preservation (Perumal, 1972). On berries that grow after harvest or out of season, damage can be severe (Fig. 5).

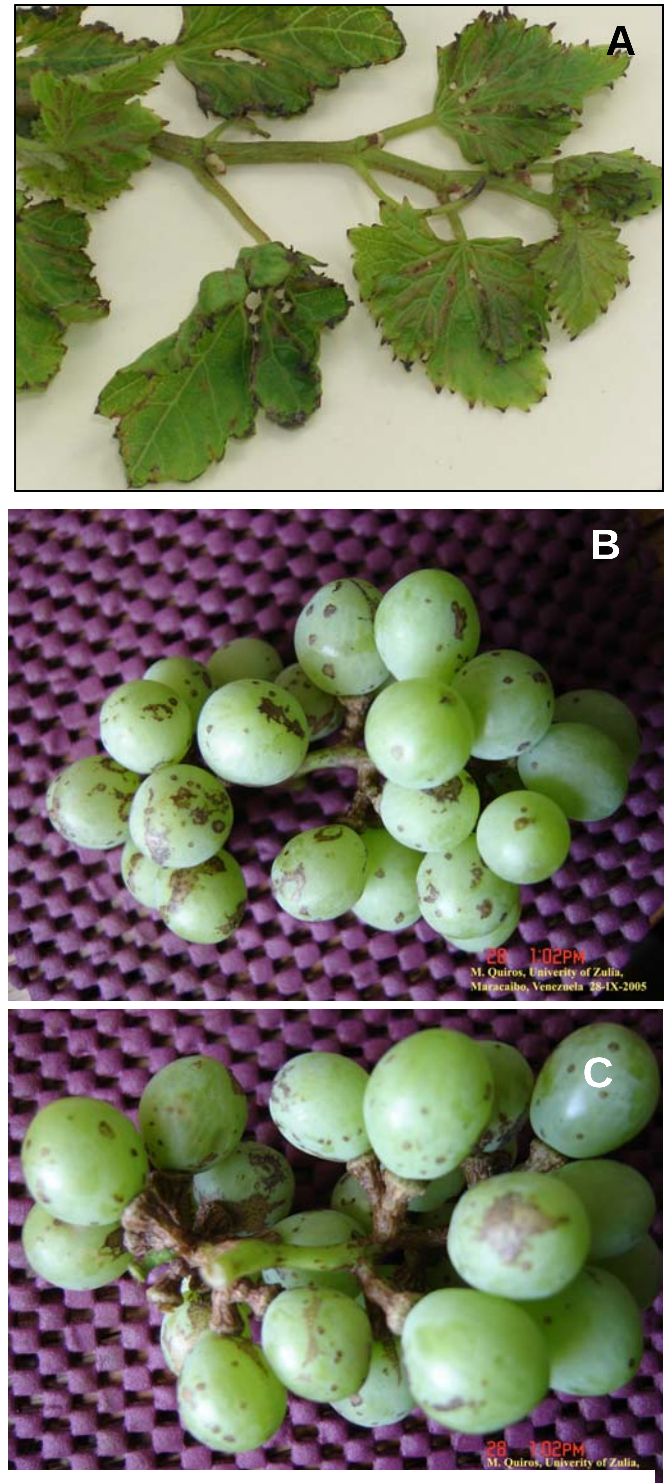

Figure 4. Damage caused by *S. dorsalis* on grape leaves and fruit Photos courtesy of Dr. Magally Quiros, University of Zulia, Maracaibo, Venezuela.

Known Hosts

S. dorsalis is highly polyphagous and has been recorded from more than 112 plant species spread across 40 different families. Its main wild host plants are thought to be various Fabaceae such as *Acacia* spp., *Brownea* spp., *Mimosa* spp., and *Saraca* spp.. Venette & Davis (2004) provide a thorough review of host plants known from the scientific literature. Recent field surveys in the Caribbean have shown that *S. dorsalis* is found on cucumbers, cantaloupe, watermelon, pumpkin and squash, hot peppers, tomato, eggplant, okra, and beans (Ciomperlik & Seal, 2004).

Major hosts:
Allium cepa (onion), *Anacardium occidentale* (cashew nut), *Arachis hypogaea* (peanut), *Camellia sinensis* (tea), *Capsicum frutescens* (chilli pepper), *Citrus* spp., *Fragaria* spp. (strawberry), *Gossypium* (cotton), *Hevea* spp. (rubber), *Hydrangea* spp., *Lycopersicon esculentum* (tomato), *Mangifera indica* (mango), *Nelumbo*

spp. (lotus), *Nicotiana tabacum* (tobacco), *Ricinus communis* (castor bean), *Rosa* spp. (rose), *Tamarindus indica* (tamarind), and *Vitis* (grape). *S. dorsalis* is only cited as a significant pest of *Citrus* in Japan and Taiwan.

Minor hosts:
Abelmoschus esculentus (okra), *Acer* spp. (maple), *Asparagus officinalis* (asparagus), *Chrysanthemum* spp., *Cucumis* spp. (cucumber and melon), *Cucurbita moschata* (pumpkin), *Dahlia* spp., *Syzygium malaccense* (malay apple), *Euonymus japonicus, Ficus carica* (common fig), *Glycine max* (soybean), *Laurus nobilis* (bay laurel), *Momordica charnatia* (balsam pear), *Musa* spp. (banana), *Pieris japonica*, *Phaseolus vulgaris* (bean), *Prunus* spp. (cherry), *Pyrus* spp. (pear), *Quercus glauca* (Japanese blue oak), *Rhododendron* spp., *Solanum melongena* (egg plant), *Virburnum* spp., and *Vigna radiata* (mung bean).

Figure 5. Severely damaged grape. Photo courtesy of Dr. Magally Quiros, University of Zulia, Maracaibo, Venezuela.

Known Vectors (or associated organisms)
S. dorsalis is also a vector of *Tomato spotted wilt viru*s (TSWV). *S. dorsalis* is also known to vector Peanut chlorotic fan-spot virus (Groundnut chlorotic fan-spot virus), Peanut yellow spot virus and *Tobacco streak virus*.

Known Distribution
Asia: Bangladesh, Brunei, China, India, Indonesia, Israel, Japan, Republic of Korea, Malaysia, Myanmar, Pakistan, Philippines, Sri Lanka, and Thailand. **Africa:** Ivory Coast and South Africa. **North America:** United States (Florida, Hawaii, and Texas). **Oceania:** Australia, Papua New Guinea, and Solomon Islands. **Caribbean:** Recently reported as an invasive species in Puerto Rico, Barbados, Jamaica, Saint Lucia, Saint Vincent, the Grenadines, and Trinidad and Tobago. **South America:** Reported in Suriname and reported causing damage to grapevine in Venezuela.

Potential Distribution in the United States
S. dorsalis was first identified from Hawaii in 1987. In October 2005, *S.dorsalis* was positively identified by USDA-ARS-SEL from specimens submitted by Palm Beach County, Florida. As of 11/30/05, *S. dorsalis* has been collected in Florida 77 times in 16 counties. The pest was primarily found on potted roses sold in retail outlets from the Florida Keys to Tallahassee. A few positive detections occurred on pepper and *Illicium* spp. Only 2 of 77 positives were in a natural environment, so it is unknown at this time if *S. dorsalis* is established in the natural environment. Subsequent to the October

detection, on 11/16/05, additional specimens were identified from Texas. Surveys in Texas have resulted in positive detections in 3 counties on *Capsicum* spp. on 11/10/05 and 11/14/05. Recently in early 2006, *S. dorsalis* was confirmed from samples collected in Jardin La Ceiba, Puerto Rico. Venette and Davis (2004) indicate that the potential geographic distribution of *S. dorsalis* in North America would extend from southern Florida to north of the Canadian boundary, as well as to Puerto Rico and the entire Caribbean region with approximately 28% of the continental United States having a suitable climate for *S. dorsalis.* Meisner et al. (2005) indicated, however, that permanent establishment would likely be limited to southern and West Coast states. A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 6) showed similar results to Meisner et al.

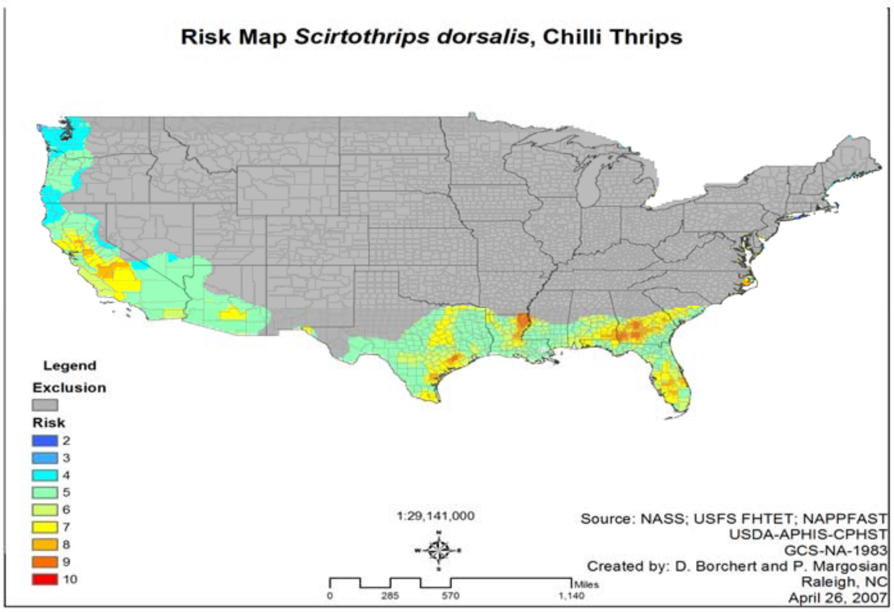

Figure 6. Risk map for *S. dorsalis* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Survey

<u>Preferred Method:</u>

Researchers typically depend on visual inspections of plant material to monitor populations of *S. dorsalis*. *S. dorsalis* feeds on new growth of nearly all vegetative plant parts (buds, leaves, flowers, fruits, and stems) (Ananthakrishnan, 1971; Chang et al., 1995). As with many thrips species, feeding deforms young leaves and stains or scars fruits (Chang et al. 1995). At the initial stage of infestation, the underside surfaces of the leaves become shiny. *S. dorsalis* is found on the leaves, flowers, and fruits of the hosts listed in this document. **Thus, malformed fruits and foliage should be examined during inspections of potential host material. However, because of their small**

size and propensity to occur inside flowers or buds, *S. dorsalis* easily could be missed during visual inspections. Visual inspections are particularly likely to be ineffective if thrips densities are low. Pupae may be found in the axils of leaves, in leaf curls, and under the calices of flower and fruits, as well as in the soil.

Suwanbutr et al. (1992) rinsed thrips from plant material using 70% ethanol and counted individuals collected on a fine muslin sieve. In Florida, 5 to 20 leaves from symptomatic plants are collected at random and placed in a ziplock bag to prevent the adults from escaping. The bag is labeled with collection locality information, host plant, date collected, and the name of collector. The samples are sent for next-day delivery to an expert for further processing to establish or confirm their identity. Foliage is washed in 70% ethanol to remove the adults and immatures. The alcohol and thrips samples are screened through a mesh diameter of 0.5 mm or less and observed under a stereomicroscope. Species level identifications should be made by a qualified taxonomist. The Florida method is recommended to enable morphological and/or molecular diagnostics to be used.

<u>Not recommended:</u> Adults may also be attracted to yellowish-green, green, or yellow boards (Tsuchiya et al., 1995). In Japan, yellow sticky traps (10 cm x 20 cm) were used to monitor *S. dorsalis* in vineyards, and these traps provided a coarse, but representative, estimate of population density on foliage (Shibao et al., 1990). A round, yellow sticky trap (15 cm diameter x 30 cm height) has also been used (Shibao et al,. 1993); traps were placed 1.2 m above the ground at a density of 4 traps per 9 m^2. Counts on yellow sticky traps are correlated with damage to grape (Shibao, 1996). Chu et al. (2006) evaluated the effectiveness of a Blue-D, CC, and sticky traps for monitoring *S. dorsalis* in Taiwan and St. Vincent. The authors recommended that a combination of visual observation, yellow sticky traps, and CC traps may be an effective *S. dorsalis* population detection and monitoring system. For CAPS surveys, yellow sticky traps are a preferred method; however, thrips specimens may be damaged in the traps, thereby hindering morphological identification. The same is true for white, blue, or yellow CC traps. When possible, whole plant specimens or thrips rinsed from specimens should be sent to diagnostic labs.

Other methods include: (1) shaking inflorescences over black paper to count nymph and adult thrips (Gowda et al, 1979); (2) dislodging thrips from young shoots on a single plant over a piece of black cardboard and counting the recovered insects (Bagle, 1993; Sureshkumar and Ananthakrishnan, 1987); (3) using sticky suction traps to monitor the flight of *S. dorsalis* (Takagi, 1978); and (4) directly counting the number of thrips on plant shoots or terminal leaves (Khanpara and Patel, 2002). However, these methods are usually used in regions where the pest is established. Due to the difficulty of identification of *S. dorsalis*, these methods are not recommended for an early detection survey.

A Berlese funnel has been used to determine the presence of thrips in bulky plant material. However, its efficiency has not been evaluated for *Scirtothrips dorsalis*. (See http://www.extento.hawaii.edu/Kbase/reports/berlese_funnel.htm for further information).

No pheromones have yet been identified for this species.

For identifiers at the ports, a shaker box technique has been developed to detect *S. dorsalis* and other actionable pests in peppers from St. Lucia and St. Vincent and was found to be superior to detecting thrips when compared to a 2% visual inspection.

For additional information on chilli thrips, including photos of symptomatic plants and chilli trips, sampling, management, and more see: http://mrec.ifas.ufl.edu/lso/thripslinks.htm).

Key Diagnostics

Identification during immature and adult stages can be done most reliably by polymerase chain reaction and restriction fragment length polymorphism (PCR-RFLP), Using a method described by Toda and Komazaki (2002) and Brunner et al. (2002).

Adult members of the genus *Scirtothrips* are readily distinguished from all other Thripidae by the following characters: surface of pronotum covered with many closely spaced transverse striae; abdominal tergites laterally with numerous parallel rows of tiny microtrichia; sternites with marginal setae arising at posterior margin; and metanotum with median pair of setae arising near anterior margin. Larvae are pale and indistinguishable from the larvae of other thrips species (CABI, 2004)

To diagnose *S. dorsalis*: Abdominal sternite with dark antecostal ridge. Ridge is not always visible for teneral adults. Lateral microtrichial fields of abdominal tergites with three discal setae. Posteromarginal comb on abdominal tergite VIII complete. Forewing shaded, lighter distally with straight cilia. Second vein: incomplete with two or three intermittent setae in distal half. Forked sense cone. Antennal segments I-II pale, III-VIIII dark. Head with three pairs of ocellar setae. Ocellar setae III between posterior ocelli. Note: the multiple transverse striae are characteristic of the genus. Posteromarginal seta II is broader and about 1.5 times longer than posteromarginal setae I and III. For images and further information on the diagnostic features of *S. dorsalis* see http://mrec.ifas.ufl.edu/lso/DOCUMENTS/Scirtothrips%2520dorsalis%2520Hood%2520ID%2520Aid.pdf

An identification system, fully illustrated with photomicrographs of structural details, together with a molecular method for distinguishing this species from related species is provided in the thrips identification tool, "*Pest Thrips of the World*" ,available for purchase at http://www.cbit.uq.edu.au/software/pestthrips/purchase.htm.

Easily Confused Pests

Figure 7. *Chaetanaphothrips orchidii* (left) and *Selenothrips rubrocinctus* (right). Photos courtesy of Lance Osbourne (http://mrec.ifas.ufl.edu/lso/thripslinks.htm) and the University of Florida, respectively.

Thrips are extremely small and very difficult to distinguish from one another, especially when immature. Adults and late-instar larvae may be identified upon close examination of morphological characters by a taxonomist (Toda and Komazaki, 2002).

Scirtothrips dorsalis has reportedly been confused with *S. aurantii* (South African citrus thrips), *S. oligochaetus,* and *Frankliniella schultzei* on various hosts (Amin and Palmer, 1985; Gilbert,1986; Toda and Komazaki, 2002).

Another similar species is *Drepanothrips reuteri*, a native European pest of grapevine, which has 6-segmented antennae (the 3 terminal segments being fused) instead of the 8-segmented found in *S. dorsalis* (CABI, 2004).

Chaetanaphothrips orchidii (orchid thrips) is sometimes confused with *S. dorsalis* in Florida (Fig. 7). This species of thrips looks very much like chilli thrips. However, there is a "break" in the dark coloring of the wings. This gives the orchid thrips the appearance of having 2 dark spots on the front portion of the abdomen. The red banded thrips, *Selenothrips rubrocinctus* (Fig. 7), causes similar damage to roses but the adult red banded thrips is black in color whereas the chilli thrips is much smaller and very light yellow, brown, or straw colored.

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Spodoptera littoralis

Scientific Name
Spodoptera littoralis Boisduval

Synonyms:
Hadena littoralis, Noctua gossypii, Prodenia littoralis, Prodenia litura, Prodenia retina, Spodoptera retina, Spodoptera 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: AHP Prioritized Pest List

Pest Description
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).

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

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, cylindrical, tapering towards the posterior and variable in color (blackish-grey 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; and 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.

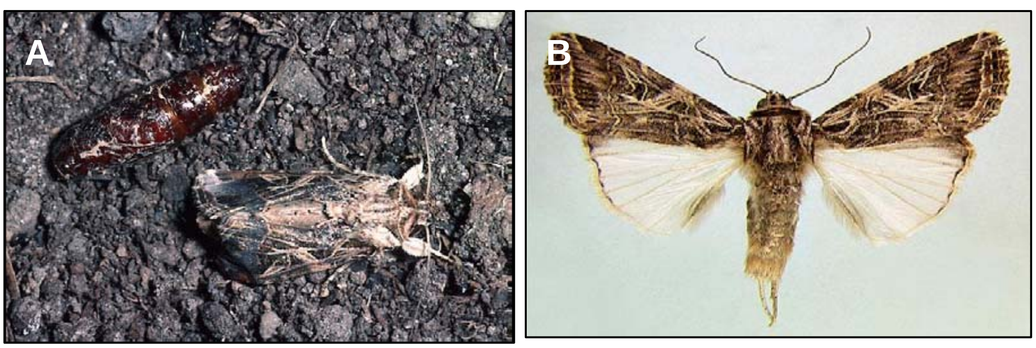

Figure 3. Pupa and adult of *S. littoralis* on soil (A). Adult moth of *S. littoralis* (museum set specimen) (B). Photos courtesy of CABI, 2004 and Entopix.

Pupae: When newly formed, pupae are green with a reddish color on the abdomen, turning dark reddish-brown after a few hours (Fig. 3). 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 grey-brown body (Fig. 3), 15 to 20 mm long; wingspan 30 to 38 mm; forewings grey 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 greyish white, iridescent with grey margins, and usually lack darker veins (EPPO, 1997).

Biology and Ecology
S. littoralis is a multivoltine species that does not enter a diapause stage. Female moths lay most of their egg masses (20-1,000 eggs) on the lower leaf surface of younger leaves or upper parts of the plant. Eggs begin to hatch after 28.6 degree days (DD) at a base temperature of 14.8 °C. The optimal temperature for egg hatch is 28-30°C. As the insect develops, it completes six instars. Early instars remain on the underside of leaves and feed throughout the day. On cotton, the first three larval instars feed mainly on the lower surface of the leaves, whereas later instars feed on both surfaces. Third and fourth instars remain on a plant, but do not feed during the daylight; later instars migrate off the plant and rest in the soil during the day and return to the plant at night. Upon pupation, the fully grown larva pushes on the loose surface of the soil downwards until it reaches more solid ground 3-5 cm deep. It then creates a clay ‘cell’ or cocoon in which it usually pupates within 5-6 hours. Emergence of adult moths occurs at night, and they have a life span of 5-10 days. Adults fly at night, mostly between the hours of 8 pm and midnight.

Pest Importance

S. 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* spp., cotton, corn, and other vegetables are affected. In Egypt, it is one of the most serious cotton pests.

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. On grape, larvae gnaw holes in the leaves until sometimes only the veins remain. The damage caused by larvae to grapevines is not merely temporary; vines may suffer so severely from exposure to intense sunlight during the summer that their development in the following year will be retarded. Larvae also gnaw at grape bunch stalks, which as a result, dry up, and the larvae feed on the grape berries (USDA, 1982).

Known Hosts

The host range of *S. littoralis* covers over 40 families, containing at least 87 species of economic importance (Salama et al., 1970).

Major Hosts

Abelmoschus escultentus (okra), *Allium* spp. (onion), *Amaranthus* spp., *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. (water hyacinth), *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)

S. 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. 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 Samoa.

Potential Distribution in 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 only as far north as Maryland (USDA, 1982). Migratory moths may be capable of periodic spread into northern states and even Canada by late summer or early fall. Venette et al. (2003) suggest that approximately 49% of the continental United States would be suitable for *S. littoralis.* A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 4) shows that portions of Arkansas, Louisiana, 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.

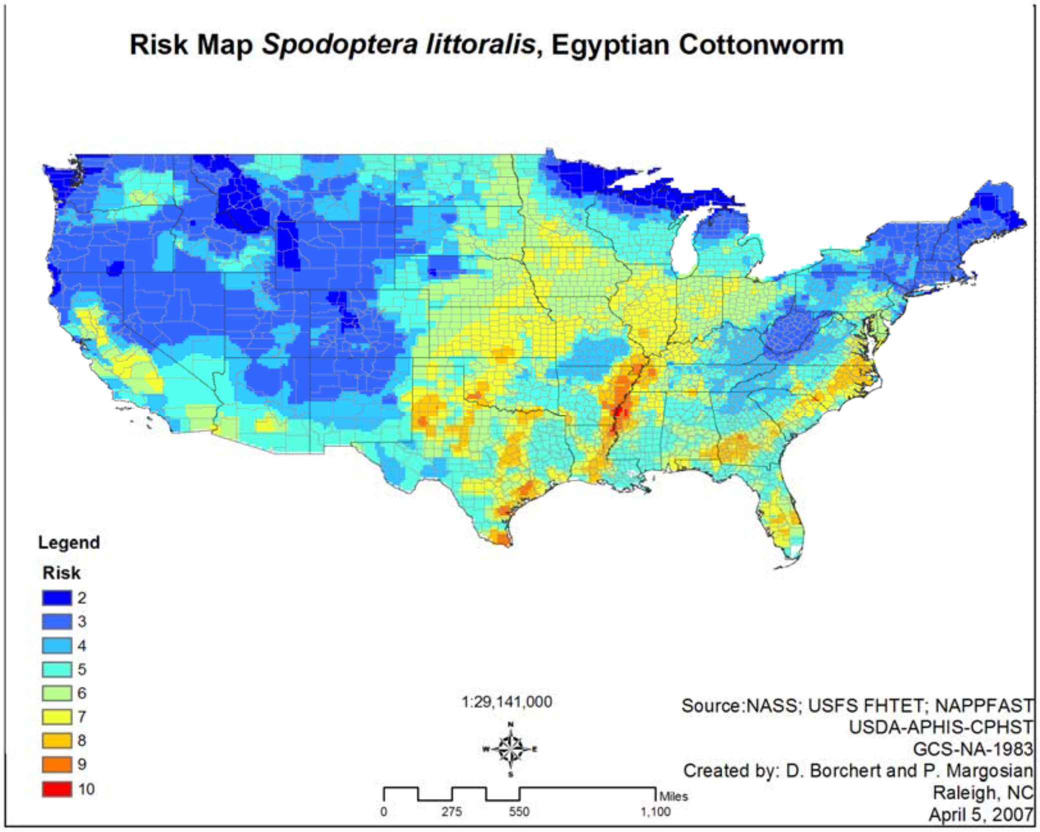

Figure 4. Risk map for *S. littoralis* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

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 CPHST- Otis lab. 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). Catches in pheromone traps did not correlate as well with densities of egg-masses in cotton fields as did catches in a black-light trap (Rizk et al., 1990). The attractiveness of traps baited with (*Z*,*E*)-(9,11)-tetradecadienyl acetate is governed primarily by minimum air temperature, relative humidity, adult abundance, and wind velocity. Densities of female *S. littoralis* also affect the number of males that are captured at different times of the year (Rizk et al., 1990). 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 United States), and *P. gossypiella* (exotic established in the United States). Lures for *S. littoralis* may also attract *Erastria* sp. (established in the United States) (PPQ, 1993).

<u>Alternative Method:</u> 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, 1968; Abul-Nasr et al.,1971).

<u>Not recommended:</u>
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).

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

Key Diagnostics
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, 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 United States, but has been reported in Hawaii.

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

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* (Fig. 6), the yellow striped armyworm, 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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http://www.aphis.usda.gov/plant_health/plant_pest_info/pest_detection/downloads/pra/slittoralispra.pdf

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.

<u>Pupa:</u> 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).

<u>Adult:</u> 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.

Biology and Ecology

The eggs of *S. litura* are laid in bunches of 50 to 300 on the under surface of leaves (preferred) by female moths (Chari and Patel, 1983). They hatch in 3 to 4 days. A single female lays 1500 to 2500 eggs in about 6 to 8 days. Castor bean is the most preferred host for ovipositing females (Chari and Patel, 1983). Newly irrigated fields are also very attractive to ovipositing females. Three peak periods of egg laying have been observed in the third weeks of June and July and in mid-August. Newly hatched larvae feed gregariously on the epidermis of the leaf. If the population density is high or the host is not suitable, the young larvae will hang on silken threads and migrate to other leaves or preferred hosts. There are generally six instars. The general habit of the larva is that the 1^{st}, 2^{nd}, and 3^{rd} instars remain on the lower surface of leaves. The 4^{th}, 5^{th}, and 6^{th} instars escape from sunshine, push to loosen the surface of the soil, and bite out soil particles to form a clay cell or cocoon in which to pupate (Chari and Patel, 1983).

Ahmed et al. (1979) showed that *S. litura* adults developed from first instar larvae in 23.4 days at 28 °C. Mean female longevity was 8.3 days and mean fecundity was 2673 eggs. Mean male longevity was 10.4 days. No mating took place on the night of emergence and maximum mating response occurred on the second night after emergence (Yamanaka et al., 1975; Ahmed, 1979). According to Yamanaka et al. (1975) the female continues to lay eggs in egg masses over a period of 5 days at 25°C.

Maximum fecundity for *S. litura* was observed at 27°C under 12 hours per 24 hours of light (100 foot candle light) (Hasmat and Khan, 1977, 1978). Temperatures between 24 and 30°C were also favorable for fecundity and fertility. At 33 and 39°C, both fecundity and fertility were decreased, and in the latter, fertility was completely inhibited (Hasmat and Khan, 1977). Twenty four hours exposure to light markedly reduced both fecundity and fertility. Hatching was highest in dark conditions (Hashmat and Khan, 1978). Parasuram and Jayaraj (1983a) noted that 25°C and 75% relative humidity were favorable for development of *S. litura* with a shorter larval period, 100% pupation, a shortened pupal period, and 100% adult emergence.

Ranga Rao et al. (1989) reported that an average of 64 degree-days (DD) above a threshold of 8°C was required for oviposition to egg hatch. The larval period required 303 DD, and the pupal stage 155 DD above a 10°C threshold. Females needed 29 DD above a 10.8°C threshold from emergence to oviposition. The upper developmental threshold temperature of all stages was 37°C; 40°C was lethal.

Maheswara Reddy (1983) showed that the majority of mating was occurred between 23.30 and 00.30 hrs under controlled conditions. The duration of matings ranged between 82.5 and 90 minutes. Although males are capable of insemination throughout their lifecycle, no males inseminated more than one female in one night. Some males failed to inseminate even one female on some nights. The mean number of mating per males was 10.3 and per female was 3.1 (Ahmed et al., 1979). Ohbayashi et al. (1973) showed two peaks in mating behavior at 23.00 (3 hours after initiation of a dark period) and a minor peak at 3:00 (1 hour before the end of the dark period).

S. litura spends its pre-pupal and pupal period inside soil. In India, Parasuraman and Jayaraj (1983b) found pupation was maximal under fallen leaves, especially in wet, sandy loam soil. Although the depth of pupation varied, no pupation was observed beyond 12 cm deep. Across soil types, most larvae pupated at a 4 cm depth.

Symptoms/Signs

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.

S. 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).

<u>Grape:</u> Larvae scrape the leaf tissue and cause ‘drying of the leaves’ (Balasubramaniam et al., 1978). The larvae damage the growing berries and cause defoliation. Balikai et al. (1999) also showed that later instar larvae cut the rachis of grape bunches and petioles of individual berries during the night hours leading to fruit drop. During the day, the larvae move to the lower portion of the leaf vines and the

crevices of the soil. The larvae use the main stem for climbing from the soil level during dusk.

<u>Other crops:</u> 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).

Pest Importance

S. litura larvae are polyphagous defoliators, seasonally common in annual and perennial agricultural systems in tropical and temperate Asia. This noctuid is often found as part of a complex of lepidopteran and non-lepidopteran foliar feeders but may also damage tubers and roots. Hosts include field crops grown for food and fiber, plantation and forestry crops, as well as certain weed species (CABI, 2004).

Most work on the economic impact of *S. litura* has been conducted in India, where it is a serious pest of a range of field crops. It has caused 12 to 23% loss to tomatoes in the monsoon season, and 9 to 24% loss in the winter (Patnaik, 1998). In a 40- to 45-day-old potato crop, damage ranged from 20 to 100% in different parts of the field depending on moisture availability. Larvae also attacked exposed tubers when young succulent leaves were unavailable (CABI, 2004). *S. litura* is also a pest of sugarbeet, with infestations commencing in March and peaking in late March and April (Chatterjee and Nayak, 1987). Severe infestations led to the skeletonization of leaves, as well as feeding holes in roots that rendered the crop 'virtually unfit for marketing'. Late harvested crops were most severely affected and, in extreme cases, 100% of the roots were damaged, leading to considerable yield reduction. Aroid tuber crops (including taro (*Colocasia esculenta*)) suffered yield losses of up to 29% as a result of infestation by *S. litura*, *Aphis gossypii* and spider mites (Pillai et al., 1993).

S. litura causes damage to many species of forest and plantation trees and shrubs (Roychoudhury et al., 1995). It is responsible for brown flag syndrome in banana (Ranjith et al., 1997) and 5 to 10% fruit damage in grapes (Balikai et al., 1999).

S. litura is also a member of a complex that causes extensive defoliation of soybean (Bhattacharjee and Ghude, 1985). Defoliation as severe as 48.7% during the pre-bloom stage of growth caused no 'marked' difference from a control treatment in which defoliation was prevented by repeated insecticide application. Number and weight of pods and grains per plant were, however, reduced when defoliation occurred at, or after, blooming.

Insecticide resistance has been reported in India (Armes et al., 1997; Kranthi et al., 2001) and Pakistan (Ahmad et al, 2007).

Known Hosts

Both *S. litura* and *S. littoralis* are widely polyphagous (Brown and Dewhurst, 1975; Holloway, 1989). The host range of *S. litura* covers at least 120 species (Venette et al., 2003). Among the main crop species attacked by *S. litura* in the tropics are taro, cotton, flax, peanuts, jute, alfalfa, corn, rice, soybeans, tea, tobacco, vegetables, aubergines eggplant), *Brassica* spp., *Capsicum* spp., cucurbits, beans, potatoes, sweet potatoes, grape, and cowpea. Other hosts include ornamentals, wild plants, weeds and shade trees (for example, *Leucaena leucocephala,* the shade tree of cocoa plantations in Indonesia). Balasubramanian et al. (1984) showed better larval growth and higher adult fecundity when reared on castor bean compared to tomato, sweet potato, okra, cotton, sunflower, eggplant and alfalfa.

Major Hosts

Abelmoschus esculentus (okra), *Acacia mangium* (brown salwood), *Allium cepa* (onion), *Amaranthus* (grain amaranth), *Arachis hypogaea* (peanut), *Beta vulgaris* var. *saccharifera* (sugarbeet), *Boehmeria nivea* (ramie), *Brassica*, *Brassica oleracea* var. *botrytis* (cauliflower), *Brassica oleracea* var. *capitata* (cabbage), *Camellia sinensis* (tea), *Capsicum frutescens* (chilli), *Castilla elastica elastica* (castilloa rubber), *Cicer arietinum* (chickpea), *Citrus*, *Coffea* (coffee), *Colocasia esculenta* (taro), *Corchorus* (jutes), *Corchorus olitorius* (jute), *Coriandrum sativum* (coriander), *Crotalaria juncea* (sunn hemp), *Cynara scolymus* (artichoke), *Erythroxylum coca* (coca), *Fabaceae* (leguminous plants), *Foeniculum vulgare* (fennel), *Fragaria ananassa* (strawberry), *Gladiolus* hybrids (gladiola), *Glycine max* (soybean), *Gossypium* spp.(cotton), *Helianthus annuus* (sunflower), *Hevea brasiliensis* (rubber), *Ipomoea batatas* (sweet potato), *Jatropha curcas* (Barbados nut), *Lathyrus odoratus* (sweet pea), *Lilium* spp. (lily), *Linum usitatissimum* (flax), *Lycopersicon esculentum* (tomato), *Malus domestica* (apple), *Manihot esculenta* (cassava), *Medicago sativa* (alfalfa), *Morus alba* (mora), *Musa* spp. (banana), *Nicotiana tabacum* (tobacco), *Oryza sativa* (rice), *Papaver* (poppies), *Paulownia tomentosa* (paulownia), *Phaseolus* (beans), *Piper nigrum* (black pepper), *Poaceae* (grasses), *Psophocarpus tetragonolobus* (winged bean), *Raphanus sativus* (radish), *Ricinus communis* (castor bean), *Rosa* (roses), *Sesbania grandiflora* (agati), *Solanum melongena* (aubergine, eggplant), *Solanum tuberosum* (potato), *Sorghum bicolor* (sorghum), *Syzygium aromaticum* (clove), *Tectona grandis* (teak), *Theobroma cacao* (cocoa), *Trigonella foenum-graecum* (fenugreek), *Vigna mungo* (black gram), *Vigna radiata* (mung bean), *Vigna unguiculata* (cowpea), *Vitis vinifera* (grape), *Zea mays* (corn), and *Zinnia elegans* (zinnia).

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

Known Vectors (or associated insects)

S. litura is not a known vector and does not have any associated organisms.

Known Distribution

The tobacco caterpillar, *S. litura*, is one of the most important insect pests of agricultural crops in the Asian tropics. This species is widely distributed throughout tropical and temperate Asia, Australasia and the Pacific Islands (Kranz et al., 1977).

Asia: Afghanistan, Bangladesh, Brunei Darussalam, Cambodia, China, Christmas Island, Cocos Islands, India, Indonesia, Iran, Japan, Korea, Laos, Lebanon, Malaysia, Maldives, Myanmar, Nepal, Oman, Pakistan, Philippines, Singapore, Sri Lanka, Syria, Thailand, and Vietnam. **Europe:** Russia. **Africa:** Reunion. **North America:** United States (Hawaii). **Oceania:** American Samoa, Australia, Belau, Cook Islands, Federated states of Micronesia, Fiji, French Polynesia, Guam, Kiribati, Marshall Islands, New Caledonia, New Zealand, Niue, Norfolk Island, Northern Mariana Islands, Papua New Guinea, Pitcairn Islands, Samoa, Soloman Islands, Tonga, Tuvalu, Midway Islands, Wake Island, Vanuatu, and the Wallis and Futuna Islands.

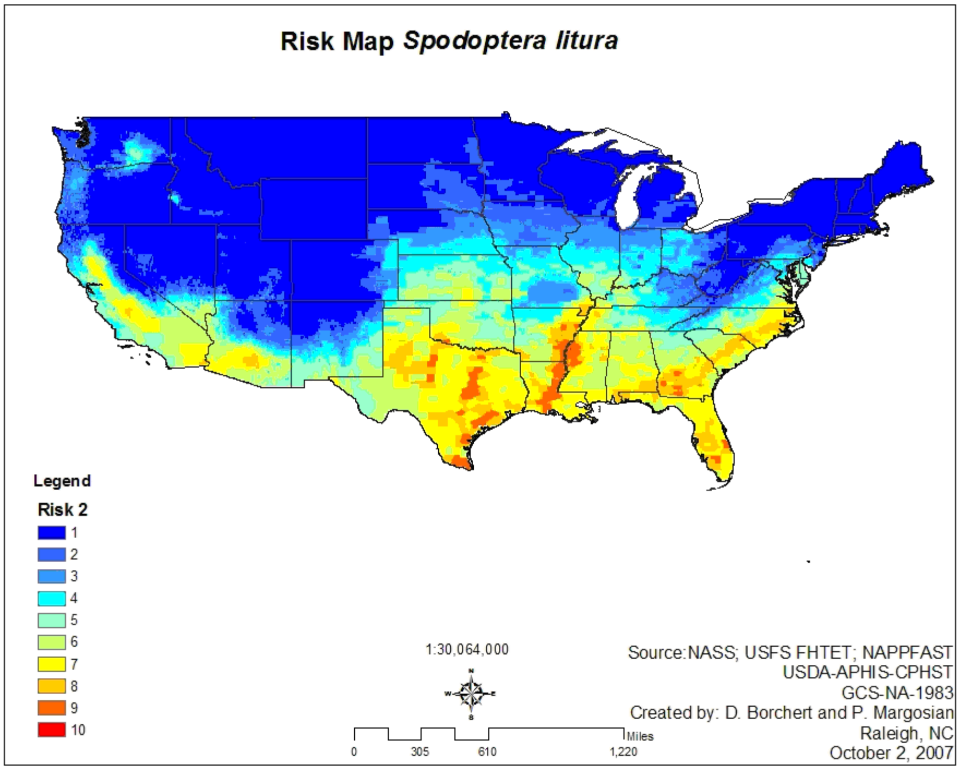

Figure 2. Risk map for *S. litura* within the continental United States. The greater the risk (climate and host availability), the greater the risk number.Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Potential Distribution within the United States

The pest has been 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. 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.

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).

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).

Key Diagnostics

Wing coloration has been used to separate the sexes of *S. litura* (Singh et al., 1975). *S. 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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Secondary Pests of Grape (Truncated Pest Datasheet)

Adoxophyes orana

Scientific Name

Adoxophyes orana Fischer von Roeslerstamm

Synonyms:
Adoxophyes reticulana, Capua reticulana, Cacoecia reticulana, Capua orana, Tortrix ornana, Tortrix reticulana, Capua congruana, Adoxopjues tripsiana, Adoxophyes fasciata, Adoxophyes congruana, Acleris reticulana.

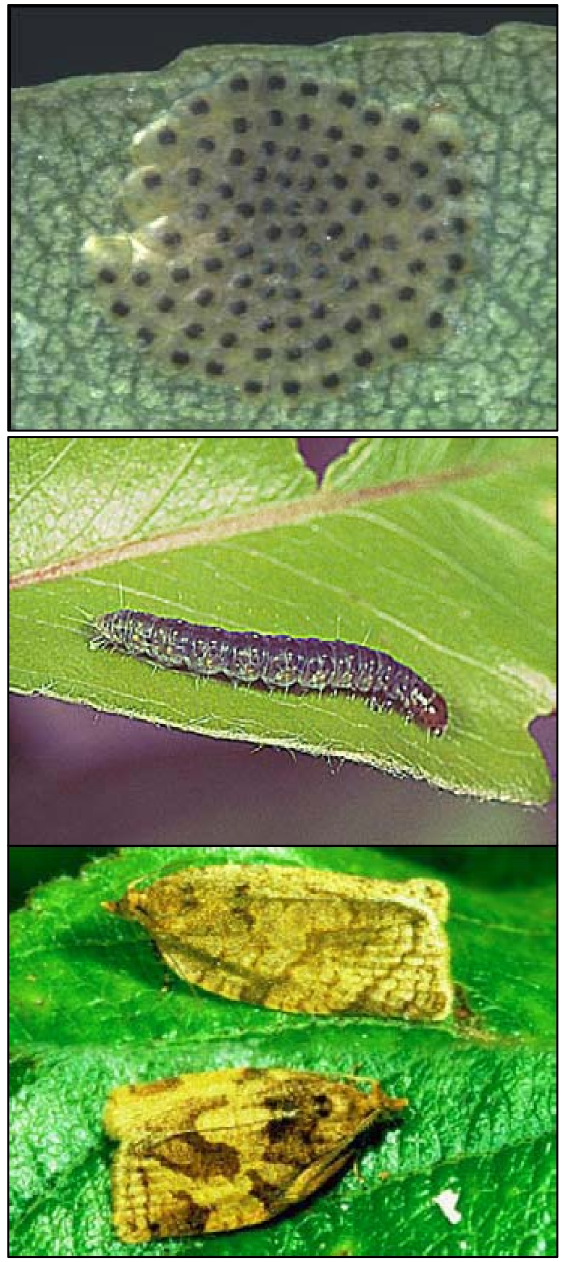

Figure 1. Eggs, larva, and adult *A. orana* (female top, male bottom). Photos courtesy of R. Coutin/OPIE.

Common Name

Summer fruit tortrix, reticulated tortrix, apple peel tortricid

Type of Pest

Moth

Taxonomic Position

Class: Insecta, **Order:** Lepidoptera, **Family:** Tortricidae

Reason for Inclusion in Manual

CAPS Target: AHP Prioritized Pest List

Pest Description

Eggs: Yellowish and deposited in masses (Fig. 1). After hatching, the transparent egg shells remain present.

Larvae: (Fig. 1) Greenish with light hairs and warts. The head is light brown to yellow (sometimes somewhat spotted) as is the thoracic shield and the anal shield. The anal comb is very fine and long with light colored teeth. The thoracal legs are brown to black. The head is long and wide. Abdominal and anal prolegs are greenish.

Pupae: The pupae of *A. orana* are initially light brown, but become dark brown towards the time of emergence of the adult moth. The length is between 8 and 11 mm. The posterior margin of abdominal segments 2 to 8 of the pupae contains very small bristles. These bristles cannot be distinguished with a regular magnifying glass and are hence visible as a line. The specific fork-shape of wing veins 7 and 8 is already visible in the pupal stage.

Adults: A very specific characteristic of *A. orana* is the fork-shaped structure of the wing veins 7 and 8. The forewing of the female is rather dull greyish brown, while in the male the coloration is brighter and is a yellowish brown (Fig. 1). Male wingspan 15-19 mm, female 18-22 mm. Sexual dimorphism pronounced; antenna of male shortly ciliate, forewing with broad costal fold from base to about one-third, markings usually conspicuous, contrasting with paler ground color; female usually larger, antenna minutely ciliate, forewing without costal fold, with darker general coloration and less contrasting markings (Bradley et al., 1973).

Figure 2. Damage to apple epidermis showing "gnawed" appearance (top) and damage to pear foliage and fruit (bottom). Photos courtesy of R. Coutin/OPIE.

Symptoms/Signs

External feeding will be visible on leaves and fresh growth of twigs. Feeding will deform leaves and create areas with necrosis (dead tissue). Leaves may appear wilted, yellow, shredded, or dead. Leaves are likely to be rolled or folded and held together with silk webbing. Feeding on new growth of twigs will leave lesions. If the insect is feeding in flowers, external feeding damage and silk webbing will be evident. In all areas where the insect has fed, frass should also be visible.

Summer generation larvae feed extensively and severely damage fruit (Fig. 2). Feeding on fruits or pods causes scabs or pitting, and frass may be present. On fruit crops, larvae prefer to feed sheltered under a leaf bound to fruit and silk.

Survey

Preferred Method: Several monitoring techniques have been developed and applied to *A. orana*. The most effective approach involves sex pheromone- baited traps. The sex pheromone is a blend of (*Z*)-9-tetradecenyl acetate and (*Z*)-11-tetradecenyl acetate. These two compounds are most attractive to males in a 9:1 blend of (Z)-9:(Z)-11 isomers; *E*-isomers of either compound had a strong inhibitory effect (Davis et al., 2005). The 9:1 pheromone blend is available commercially as Adoxomone (Murphy Pherocon™ Summer Fruit Tortrix Moth Attractant) for use with Pherocon 1C traps [Zoecon Corp]. **The lure in a 9:1 blend is available from the CPHST- Otis lab.**

<u>Alternative Method:</u> Visual sampling and beat sampling may also be used to inspect plants for eggs and larvae. Eggs may be observed on the stems and leaves; late instars may be found in the crown on new shoot growth; and pupal cocoons may be found in leaves, on stems, or in mummified pods/seeds. Both methods are time consuming. Visual sampling or beat sampling are not commonly recommended (Davis et al., 2005).

<u>Not recommended:</u> As an alternative to pheromone traps, Robinson light traps with 125W mercury vapor bulbs, 125 W black light bulbs, or 100W flood lights can be used. While sex pheromone traps attract males of a targeted species, light traps non-selectively draw in many flying insects.

Surveys should be focused where the greatest risk for establishment occurs. A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 3) indicates that most states in the United States have a risk rating of 4 or greater for *A. orana* establishment based on host availability and climate within the continental United States.

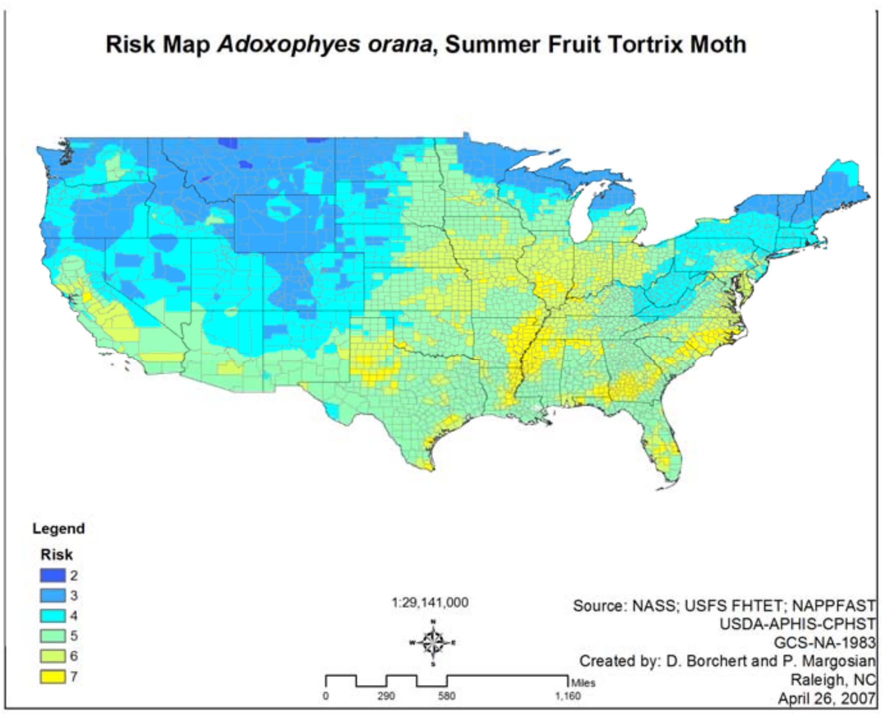

Figure 3. Risk map for *E. postvittana* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Key Diagnostics

Adoxophyes orana may occur in mixed populations with closely related or morphologically similar species. Because of their very secretive nature, leafrollers are difficult to detect. Distinguishing between males and females of adult *Adoxophyes* is difficult in general (Balachowsky 1966). According to Yasuda (1998), the extensive color and pattern variation of the forewing and morphological resemblance among *Adoxophyes* species have created difficulties in the identification of the species. *A. orana* very closely resembles two U.S. species, *Adoxophyes furcatana* and *A. negundana*, but there are slight differences in male genitalia. Any identification should be confirmed by an appropriately trained entomologist.

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Aleurocanthus spiniferus

Scientific Name
Aleurocanthus spiniferus Quaintance

Synonyms:
Aleurocanthus citricolus, Aleurocanthus rosae, Aleurocanthus spiniferus var. *intermedia*
Aleurodes citricola, Aleurodes spinifera

Common Name
Orange spiny whitefly, citrus mealy wing

Type of Pest
Whitefly

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

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

Pest Description
Eggs: The elongate-oval eggs (0.2 mm long) are yellow when first laid and then darken to charcoal grey or black; each is attached to the leaf by a short pedicel.


Figure 1. *A. spiniferus* eggs and nymphs. Nymphal instars 1, 2, and 4 (pupae, 1.2 mm in length). The white, waxy filaments are typical of the species. Photo courtesy of CABI, 2004.

Larvae:
The six-legged, dusky, elongate first-instar larvae (0.3 x 0.15 mm) have two long and several shorter, slender dorsal glandular spines. All subsequent immature stages are sessile, have non-functional leg stubs, and possess numerous, dark dorsal spines on which a stack of exuviae of earlier instars may occur. The second instar (0.4 x 0.2 mm) is a dark brown to charcoal convex disc with yellow markings, while the third instar (0.87 x 0.74 mm) is usually black with a rounded, greenish spot on the anterior part of the abdomen and obvious dorsal spines. In the fourth immature stage or 'pupa', females are larger (1.25 mm long) than males (1 mm long). This stage is black, has numerous dorsal spines, and is often surrounded by a white fringe of waxy secretion (Fig. 1). **This is the stage required for identification purposes.**

<u>Adults</u>: Winged; the females (1.7 mm long) are larger than the males (approximately 1.33 mm long). The wings are dark grey at ecdysis (Fig. 2), sometimes developing a metallic blue-grey sheen later; lighter markings on the wings appear to form a band across the insect. The body is orange to red initially; the thorax darkens to dark grey in a few hours. The limbs are whitish with pale yellow markings.

Figure 2. Adult *A. spiniferus*. Photo courtesy of M.A. van den Berg. www.invasive.org

Symptoms/Signs

Inspect for spiral egg masses and larvae on the underside of leaves. The colorful adult may be found on tender terminal growth (USDA, 1982). Sticky honeydew deposits accumulate on leaves and stems and usually develop black sooty mold fungus (Fig. 3), giving the foliage (even the whole plant) a sooty appearance. Ants may be attracted by the honeydew. Infested leaves may be distorted. The insects are most noticeable as groups of very small, black spiny lumps on leaf undersides.

Figure 3. Sooty mold on citrus leaves. Photo courtesy of M.A. van den Berg. www.invasive.org

Survey

<u>Preferred Method:</u> *Aleurocanthus spiniferus* is most often found on citrus and roses. Examine plants, especially shrubs or trees, closely for signs of sooty mold or sticky honeydew on leaves and stems, or for ants running about. A heavy infestation gives trees an almost completely black appearance. Look for distorted leaves with immature stages of *A. spiniferus* on the undersides. The adults fly actively when disturbed. Good light conditions are essential for detection; in poor light, a powerful flashlight is helpful. A large hand lens may be necessary to aid in recognition of the dorsal spines on immature stages (CABI, 2004).

Key Diagnostics

Several similar species of *Aleurocanthus* also occur on citrus, including *A. citriperdus* and *A. woglumi*. These species differ from each other only in microscopic characters of the 'pupa' and require expert preparation and identification to distinguish them reliably. The main field characteristic difference between orange spiny whitefly and citrus blackfly, *A. woglumi*, is that the white wax fringe that surrounds their pupal case margins is generally twice as large for the orange spiny whitefly.

References

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USDA. 1982. Pests Not Known to Occur in the United States or of Limited Distribution, No. 14: Orange spiny whitefly.

Eudocima fullonia

Scientific Name
Eudocima fullonia Clerck

Synonyms:
Othreis fullonia, Noctua dioscoreae, Ophideres fullonia, Ophideres obliterans, Ophideres princeps, Othreis phalonia, Othreis pomona, Phalaena fullonica, Phalaena fullonica, Phalaena phalonia, Phalaena pomona

Common Names
Fruit piercing moth, fruit-sucking moth

Type of Pest
Moth

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

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

Pest Description
Eggs: Hemispherical, just over 1 mm in diameter, and greenish-white to creamy-yellow when laid. Delicate surface sculpturing can be seen with the aid of a microscope. The brownish head capsule of the developing larva becomes obvious beneath the shell a few hours before hatching.


Figure 1. Larva of *E. fullonia*. Photo courtesy of CABI, 2004.

Larvae: The newly hatched larvae are 4-5 mm long, a bright translucent green in color, and inconspicuously banded by brown spots and hairs. The head capsule is 0.5 mm wide. Second instars are a uniform dull black, with two developing, paired, lateral orange eyespots. Larvae molt four or five times during development. Final instars can reach about 60 mm in length, with a head capsule of 4.5 mm. Mature larvae are a velvety brown to black (Fig. 1) or pale yellow to green. There are fine powdery white spots along the entire length of the body and two conspicuous, paired, lateral eyespots on the second and third abdominal segments. In dark larvae, the eyespots are peripherally white (above) and orange (below), with a central black area surrounding a pale blue core. When resting, larvae hold the posterior part of the body upwards, while the anterior part is curled with the head tucked under (Fig. 1). If disturbed, larvae may rear and 'spit' digestive juices. Larvae move with a semi-looping action.

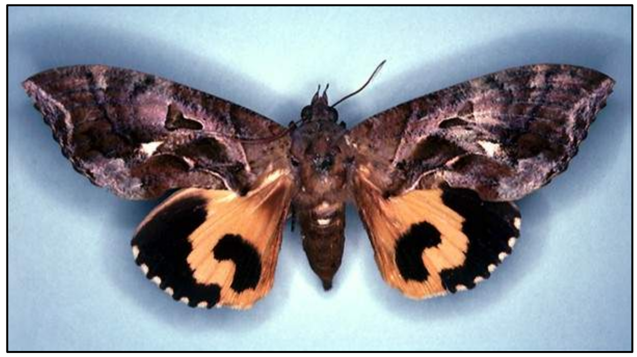

Figure 2. Adult female fruit piercing moth. Photo courtesy of CABI, 2004.

Pupae: The post-feeding larva forms a silken cocoon among the leaves of the larval host plant and attaches itself within the cocoon at the anal end. The pupa is about 30 mm long, a glistening brown-black, and can be sexed at this stage using differences in the position of the genital grooves.

Adults: Adults have 7-10 cm wingspans, with mottled brown, grey, green, or silvery white forewings. The color patterns of the forewings are sexually dimorphic. Males have leaf-like forewings of red-brown to purplish-brown. There is an inconspicuous, irregular spot centrally placed near the anterior margin. In females, the forewings are more variegated and striated than in males. The color varies between purplish-brown and greyish-ochre, often flecked with green and white. There is a distinct dark, roughly triangular mark in a similar position to the spot in the male forewing. The hind wings are characterized by the orange-yellow color, extensively bordered by a black and hatched area and a central black mark (kidney shaped or round). These are often exposed when the moth is feeding. The thorax is a purplish-brown and the abdomen orange-yellow. An individual moth can spend several hours feeding from the one fruit, but would generally attack a number of fruit on a single night. Adults rest with the forewings held tent-like over the body. When feeding, the forewings are held out exposing the bright hindwings (Fig. 3).

Figure 3. Male (left) and female (right) *E. fullonia* on a green mandarin fruit. Photo courtesy of CABI, 2004.

Symptoms/Signs

For most moth pests, the larvae are the damaging stage. **The fruit piercing moth differs in this aspect, because it is the adult moth that is the damaging stage, and the larvae are essentially not harmful.** The mouth parts of the moth are about an inch (2.5 cm) long and strong enough to penetrate through tough-skinned fruit. Once the moth has punctured the skin of the fruit, a process that usually takes a few seconds, it feeds upon the juices of the fruit (Fig. 3). Feeding occurs at night and the fruit does not have to be ripe to be fed upon by this moth. Fruit flesh damaged by this moth becomes soft and mushy differing from fruit damaged by fruit flies, which is more liquid.

Figure 4. A damaged fruit showing fruit rot around the piercing moth feeding site. Photo courtesy of CABI, 2004.

A round, pinhole-sized puncture is made in fruits. The hole serves as an entry point for pathogens and can result in early fruit drop. The latter is an obvious sign of fruit piercing moth activity in citrus. A small cavity is left in the fruit in the feeding site. The area of the fruit around the cavity will be dry and spongy. The fruit piercing moth is a known vector of *Oospora citri*, a fungus that rots the fruit and has a penetrating odor that attracts this moth. Other microorganisms that gain entrance into the fruit and cause rotting include *Fusarium* spp., *Colletotrichum* spp., and several types of bacteria. When moths are abundant, green fruit is attacked, causing premature ripening and dropping of fruits. On oranges, a green fruit turns yellow at the site of the piercing and fungi soon develop within the wound.

Survey

Preferred Method: Adult moths are likely to be found on mature fruit several weeks before harvest. The most effective way to monitor for fruit piercing moths is to inspect the crop by flashlight after sundown beginning a few weeks before harvest (Davis et al, 2005). Moths are most active in the first few hours of the night. The large, red-glowing eyes of the moths are easily seen. Check trees/vines in the two outer rows of an orchard, particularly on the leeward side. Most damage occurs in the peripheral rows. Surveys were typically initiated 30 minutes after sundown and lasted one hour.

No pheromones or semiochemicals have been identified for *E. fullonia*.

Foliage of host plants may be inspected for larvae and other life stages (Davis et al. 2005).

In some fruits, such as lychees, detection of fruit piercing moth damage can be difficult. The slightest sign of weeping can be an indication, and when the fruit is squeezed, the juice will squirt out. The damage site will be flaccid and flattened in appearance and lack the firm, rounded flesh of intact fruit. Many farmers opt to place freshly picked fruit in a cool store at high humidity, which facilitates detection of damage after one day.

Key Diagnostics

Adults of E. fullonia closely resemble species such as *Eudocima homaena* and *Eudocima jordani*. All species of *Eudocima* cause similar damage. Separation of species involves detailed microscopic examination. Fruit discoloration could be attributed to fruit fly damage, but the size of the hole left at the damage site will clarify whether fruit-piercing moths were involved.

References

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Eutetranychus orientalis

Scientific Name
Eutetranychus orientalis Klein

Synonyms:
Anychus orientalis, Anychus ricini, Eutetranychus monodi, Eutetranychus sudanicaus, Eutetranychus anneckei, Anychus latus, Eutetranychus latus

Common Name(s)
Citrus brown mite, oriental mite, oriental red mite, oriental spider mite

Type of Pest
Mite

Taxonomic Position
Class: Arachnida, **Order:** Acarina, **Family:** Tetranychidae

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

Pest Description
Eggs: The eggs of *E. orientalis* are oval or circular (Fig. 1) and flattened, coming to a point dorsally, but lacking the long dorsal stalk of other spider mites. Newly laid, the eggs are bright and hyaline, but later they take on a yellow, parchment-like color (Smith-Meyer, 1981).

Figure 1. Eggs (left) and adult (right) of *E. orientalis*. Photos courtesy of Pedro Torrent Chocarro.

Larvae: Average size of the larva of *E. orientalis* is 190 x 120 µm. The protonymph is pale-brown to light-green, with legs shorter than the body, average size 240 x 140 µm. The deutonymph is pale-brown to light-green, average size 300 x 220 µm.

Adults: Adult female *E. orientalis* are broad, oval and flattened. They vary in color from pale brown through brownish-green to dark green with darker spots within the body. The legs are about as long as the body and arevyellow-brown (Fig. 1). Average size is 410 x 280 µm. Adult male *E. orientalis* are much smaller than the females. They are elongate and triangular in shape with long legs (leg about 1.5 x body length). The body setae are short and cannot be seen with a 10x lens (Dhooria and Butani, 1984; Smith-Meyer, 1981).

Symptoms/Signs

E. orientalis begins feeding on the upper side of the leaf along the midrib and then spreads to the lateral veins, causing the leaves to become chlorotic. Pale yellow streaks develop along the midrib and veins initially, which later progress to a greyish or silvery appearance of the leaves. When damaged, the younger, tender leaves show margins that are twisted upwards. Little webbing is produced but is possible. In heavier infestations, the mites feed and oviposit over the whole upper surface of the leaf. Very heavy infestations on citrus cause leaf fall and die-back of branches, which may result in defoliated trees. Lower populations in dry areas can produce the same effect.

Survey

Preferred Method: The presence of *E. orientalis* can be detected by discoloration of the host leaves and pale-yellow streaks along the midribs and veins. Eggs, immatures stages, and adults may be observed visually on the upper leaf surface. Adult females are larger than the males. They are oval and flattened and are often pale brown through brownish-green to dark green. Webbing is possible (often dust colored), providing protection for the eggs. The spread of the mite is windborne, and new infestations commonly occur at the field perimeters. Field perimeters should, therefore, be scouted, especially field perimeters facing prevailing winds. Studies indicate that alfalfa plays a role in dispersing tetranychid mites to other crops (Osman, 1976). Fields near alfalfa should be targeted for survey. Shake leaves above white paper or cloth, and use a hand lens to observe mites.

Key Diagnostics

According to a NAPPO pest alert, the only form of *E. orientalis* that can be identified is the adult male. Conflicting information states that identification of *E. orientalis* requires examination of cleared and mounted female specimens by transmitted light microscopy. Mite experts agree that though it may be possible to identify a specimen with a slide mounted female, you can never be 100% sure without a male for confirmation. *E. orientalis* can be easily mistaken for the Texas citrus mite (*E. banksi*). Similarity of the female *E. orientalis* with other tetranychid mites such as the two-spotted mite *(Tetranychus urticae)* can make identification difficult.

References

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Smith-Meyer, M.K.P. 1981. Mite pests of crops in southern Africa. Science Bulletin, Department of Agriculture and Fisheries, Republic of South Africa, (No. 397):65-67.

Oxycarenus hyalinipennis

Scientific Name
Oxycarenus hyalinipennis Costa

Synonyms:
Aphanus hyalinipennis, Aphanus tardus var. *hyalipennis*

Common Name(s)
Cotton seed bug, cotton stainer, dusty cotton stainer, dusky cotton bug, dusky cottonseed bug, Egyptian cotton seed bug

Type of Pest
Bug

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

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

Pest Description
Eggs: Oval 0.28 x 0.95 mm, longitudinally striated, pale yellow becoming pink.

Nymphs: Head and thorax brownish-olivaceous, abdomen pinkish. Fifth instar darker brown on head and thorax, wingpads distinct, extending to at least third abdominal segment.


Figure 1. Adult *O. hyalinipennis*. Photo courtesy of Georg Goergen/IITA Insect Museum, Cotonou, Benin. CABI (2004).

Adults: (Fig. 1) Newly emerged individuals are pale pink, but rapidly turn black. Length of male about 3.8 mm; female 4.3 mm. Male abdomen terminates in round lobe, while the female's is truncate. The insects have three tarsal joints and a pair of ocelli. Second antennal segments are usually in part pale yellow. Hemelytra hyaline and usually whitish; clavus, base of corium, and costal vein more opaque than rest. Setae of 3 different types: More or less erect, stiff setae, which are blunt at tip and terminate in 4-7 small teeth; normal, straight, tapering setae; and very thin, curved, flat-lying setae (USDA, 1983).

Oxycarenus hyalinipennis begins feeding, mating, and egg laying when the seeds of its host become available. Resting adults leave their shelters, move to young cotton plants, and wait for the bolls to ripen. Females lay eggs in the lint of the open bolls. Adults and nymphs generally feed on the seeds of plants in the family Malvaceae. The last generation undergoes aestivation until seed material is available the next growing season (NPAG, 2003).

Symptoms/Signs

O. hyalinipennis has been observed sucking the fruits of grapes. The feeding damage appeared as greasy spots that exuded light colored gum. Black feces were also present on the fruit (Avidov and Harpaz, 1969).

On cotton, the lint in which the bugs have been present is stained pinkish, sometimes with a trace of green, and contaminated with crushed fragments of the insect. Cotton seeds appear undamaged on the outside; internally, the embryos are shriveled and discolored (USDA, 1983).

Survey

O. hyalinipennis has been intercepted a few times each year at U. S. ports of entry. All interceptions occurred at airports, mostly in baggage; no interceptions were recorded from preferred malvaceous hosts. These interceptions point to the risk of *O. hyalinipennis* moving on commodities that are not its hosts (NPAG, 2003).

<u>Preferred Method: C</u>urrently, there is no literature available on survey for *O. hyalinipennis* specific to grape. Visual inspection is the only survey method available at this time. Samy (1969) observed adult clusters on leaves of mango, guava, and citrus. For cotton, cotton bolls can be tapped or torn open and examined for evidence of *O. hyalinipennis*. Additionally, sweep netting of weeds between cotton rows or field edges is recommended. Adults prefer crevices in such resting sites as tree trunks, undersides of leaves on trees, pods of legumes, dried flower heads, roots of grasses, under sheath leaves of corn and sugarcane, telephone poles or wooden posts, old nests of *Polistes* spp. (paper wasps), and crevices between strands of barbed wire (Kirkpatrick, 1923). Areas of greatest risk should be targeted (Fig. 2).

Key Diagnostics

The key diagnostic involves morphological examination of adults.

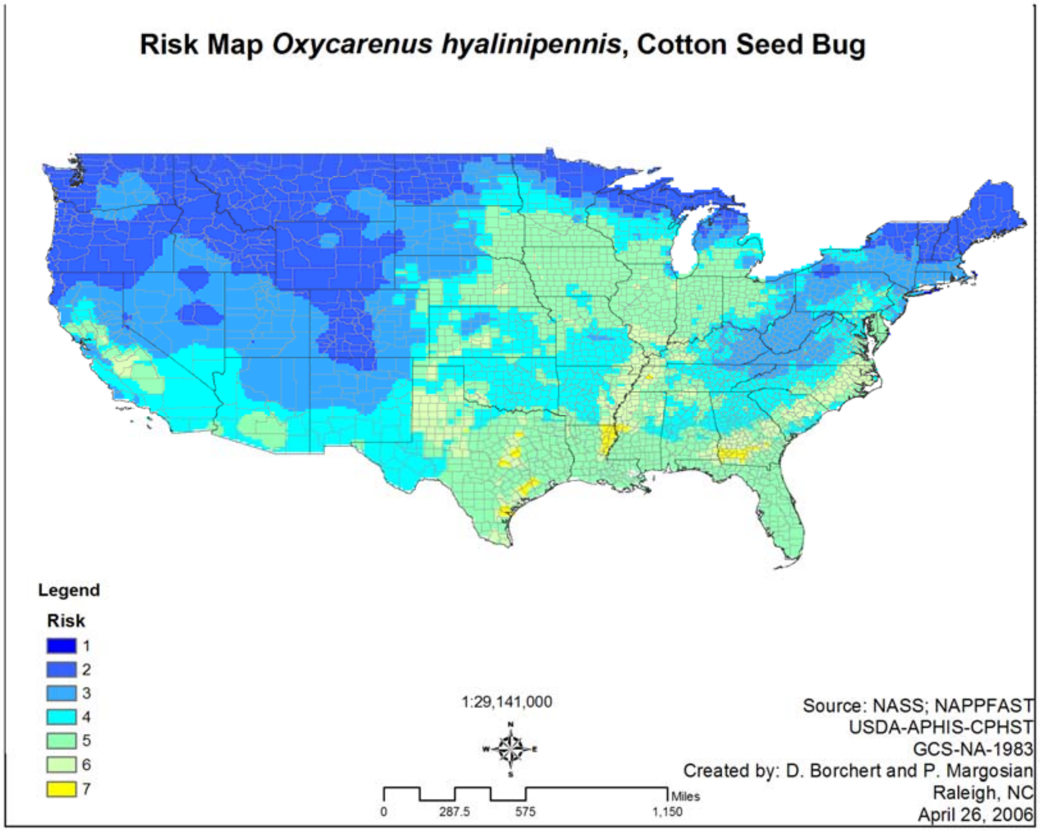

Figure 2. Risk map for *O. hyalinipennis* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

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Avidov, Z., and Harpaz I. 1969. Plant Pests of Israel. Jerusalem, Israel University Press.

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New Pest Advisory Group (NPAG). 2003. *Oxycarenus hyalinipennis* (Costa): Cotton Seed Bug. USDA-APHIS-PPQ-CPHST.

Samy, O. 1969. A revision of the African species of *Oxycarenus* (Hemiptera:Lygaeidae). Royal Entomol. Soc. London Trans. 121(4):79-165.

USDA. 1983. Pest not known to occur in the United States or of limited distribution, No. 38: Cottonseed bug.

Planococcus lilacinus

Scientific Name
Planococcus liliacinus Cockerell

Synonyms:
Dactylopius coffeae, *D. crotonis, Planococcus crotonis, P. deceptor, P. tayabanus*, *Pseudococcus coffeae, P. crotonis, P. deceptor*, *P. lilacinu*s, *P. tayabanus*, *Tylococcus mauritiensis*

Common Name(s)
Oriental mealybug, cacao mealybug, coffee mealybug

Type of Pest:
Mealybug

Taxonomic Position
Class: Insecta, **Order:** Homoptera, **Family:** Pseudococcidae

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

Pest Description
Descriptions of the egg, nymph, male pupa, and adult male are lacing except for a brief description of the life history by van der Goot (1917).

Figure. 1. *P. lilacinus* (2.7 mm) on cocoa pod. Photo courtesy of CABI, 2004.

Adult females: In the field, the adult females of *P. lilacinus* may be easily distinguished from *P. citri* by the much more globose shape of *P. lilacinus* (Fig. 1, Fig. 2). Beneath the pink to purple wax coating, the body is yellowish. The mid-dorsal line is fairly wide but indistinct. Illustrations of external features are available in Le Pelley (1968).

The mounted female is broadly oval to rotund, length 1.2 to 3.1 mm, width 0.7 to 3.0 mm. Margin of body with a complete series of 18 pairs of cerarii, usually all with stout conical setae. Legs stout: hind trochanter + femur 210 to 315 µm long, hind tibia + tarsus 210 to 275 µm long, ratios of lengths of hind tibia + tarsus to hind trochanter + femur 0.77 to 0.97; translucent pores present on hind coxae and tibiae. The inner edges of ostioles are strongly sclerotized. Circulus large and quadrate, width 105 to 200 µm.

Cisanal setae noticeably longer than anal ring setae. Anal lobe cerarii each situated on a moderately sized, well-sclerotized area (Cox, 1989).

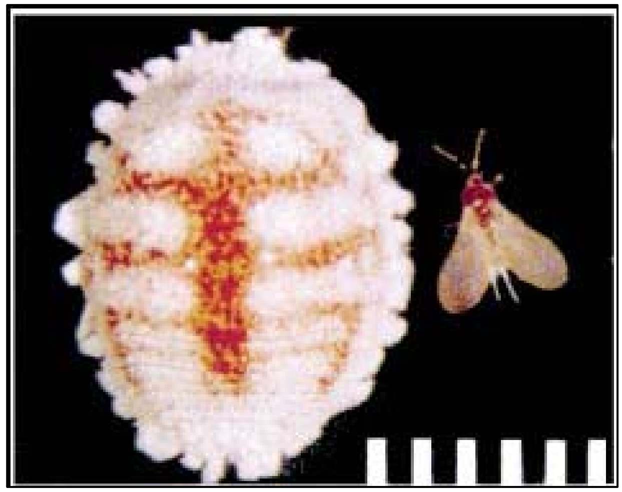

Figure 2. Adult female *P. lilacinus*. (left); winged male (right). From Kantheti, 1994; original photograph by Dr K. S. Jayarama). Each unit in the scale represents 0±5 mm.

Venter: Multilocular disc pores occurring on median area only, present around vulva as single or double rows across posterior borders of median areas of abdominal segments IV to VII and usually in a single row across anterior edge of segment VII (although the latter is sometimes reduced to a few pores). A few pores are sometimes present on anterior edges of median areas of abdominal segments V and VI.

Dorsum: Multilocular disc pores and tubular ducts absent. Setae very long, stout, and flagellate (a character which distinguishes *P. lilacinus* from many other *Planococcus* species). Length of longest setae on abdominal segments VI or VII is 50 to 140 µm.

Symptoms/Signs

There is very little information on the symptoms of attack by *P. lilacinus* in the available literature, although it has been said to cause severe damage or is listed as being a serious or the main pest of coffee, tamarind, custard apples, coconuts, cocoa, and citrus (CABI, 2004).

Figure 3. *P. lilacinus* on mango fruit. Photo courtesy of Mango Information Network.

Symptoms on coconuts and cocoa are described as button nut shedding, drying up of inflorescence, and the death of tips of branches. Dense colonies form conspicuous patches on fruits; copious honeydew excretion may result in sooty mold development near colonies and the attraction of attendant ants. Fruits have been reported to have an abnormal shape and drop prematurely.

Survey

In the field, *P. lilacinus* may be detected by thoroughly inspecting its normal habitats such as fruits, growing plant tips, shoots, and roots (CABI, 2004). On plants such as pomegranates, *Annona* spp., and *Citrus* spp., infestations on fruits, which tend to be heavy, can easily be detected (Fig. 3). Quarantine procedures should include thorough inspection of fruits, plant parts, and seedlings of suspect host plants using a hand lens.

Males were said to pupate on the underside of leaves and to be scarce (van der Goot, 1917).

On cocoa, *P. lilacinus* is attended by several ant species, including *Dolichoderus bituberculatus* (commonly referred to as the black ant) in Java and *Oecophylla longinoda*, *Technomyrmex detorquens* and *Odontomachus haematodus* in Sri Lanka. In the Philippines, *P. lilacinus* is attended by *Anoplolepis longipes [Anoplolepis gracilipes*], an ant which on cocoa in Java tends to displace *D. bituberculatus*.

Key Diagnostics

Reliable identification requires detailed study of slide-mounted adult females. *P. lilacinus* has often been mistaken in the field for other *Planococcus* spp. on cocoa and coffee, such as *P. citri* and *P. kenyae*, because of its yellowish body color beneath the wax and the presence of a median dorsal stripe extending from the first thoracic to the mid-abdominal region. It can, however, be distinguished by its more globose body shape and by having a much wider, but indistinct, median dorsal stripe.
The slide-mounted female is also quite distinct by the combination of stout legs, long dorsal setae and reduced number of multilocular disc pores, which occur mainly in small numbers in the posterior abdominal segments. A Lucid tool on mealybugs has been recently developed (see http://www.sel.barc.usda.gov/ScaleKeys/ScaleInsectsHome/ScaleInsectsMealybugs.html).

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Thaumatotibia leucotreta

Scientific Name
Thaumatotibia leucotreta Meyrick

Synonyms:
Cryptophlebia leucotreta

Common Name(s)
False codling moth, citrus codling moth, orange codling moth

Type of Pest:
Moth

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

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

Pest Description


Figure 1. Larvae of *T. leucotreta.* Photo courtesy of T. Grove and W. Styn. www.bugwood.org

False codling moth (FCM), *T. leucotreta,* is a pest of economic importance to many crops throughout sub-Saharan Africa, South Africa and the islands of the Atlantic and Indian Oceans (Stibick, 2006). The FCM is an internal fruit feeding tortricid that does not undergo diapause and may be found throughout the year in warm climates on suitable host crops. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop. *T. leucotreta* is a generalist with respect to host plant selection and has been recorded as feeding on over 50 different plant species. The generalist feeding strategy enables survival in marginal conditions as is necessary due to lack of diapause. Important host crops include avocado (*Persea americana*), citrus (*Citrus* spp.), corn (*Zea mays*), cotton (*Gossypium* spp.), macadamia (*Macadamia* spp.), and peach and plum (*Prunus* spp.) (USDA, 1984; Stibick, 2006).

Eggs: Eggs are flat, oval (0.77 mm long by 0.60 mm wide) shaped discs with a granulated surface. The eggs are white to cream colored when initially laid, then changing to reddish color before the black head capsule of the larvae becomes visible under the chorion prior to eclosion (Daiber, 1979a).

Larvae: First instar (neonate) larvae approximately 1 to 1.2 mm in length with dark pinacula giving a spotted appearance, fifth instar larvae are orangey-pink, becoming more pale on sides and yellow in ventral region, 12 to 18 mm long, with a brown head capsule and first thoracic segment (Fig. 1). The last abdominal segment bears an anal comb with 2 to 7 spines. The mean head capsule width (mm) for the first through fifth instar larvae has been recorded as: 0.22, 0.37, 0.61, 0.94 and 1.37, respectively (Daiber, 1979b).

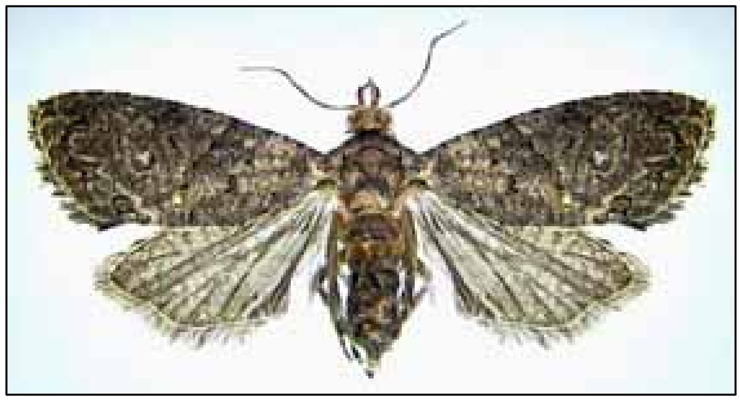

Figure 2. Adult false codling moth. Photo courtesy of CABI, 2004.

Pupae: Prepupa and pupa form inside a lightly woven silk and soil cocoon formed by the fifth instar larvae on ground. Length is 8 to 10 mm and sexual determination through morphological differences on pupal case is possible (Daiber, 1979c)

Adult: Adult body length 6 to 8 mm, wingspan of female and male moth is 15 to 20 and 15 to 18 (mm), respectively. Body brown, thorax with posterior double crest. Forewing is a mixture of plumbeous, brown, black, and ferruginous markings, most conspicuous being blackish triangular pre-tornal marking and crescent-shapted marking above it, and minute white sport in discal area. Male is distinguished from female by its large, pale grayish genital tuft, large dense grayish white brush hindlegs, and its heavily tufted hind tibia (Gunn, 1921; Couilloud, 1988; CABI, 2004).

Symptoms/Signs

In general, the habit of internal feeding by FCM larvae (Fig. 3A) displays few symptoms. Emerging larvae bore into the albedo and usually feed just below the fruit surface. Cannibalism among young larvae ensures that usually only one caterpillar matures in each fruit. When full-grown the larvae bore their way out of the fruit to seek a site for pupation. The rind around the point of infestation takes on a yellowish-brown color as the tissue decays and collapses. Larval feeding and development can affect fruit development at any stage, causing premature ripening and fruit drop.

Grape: Fresh larval penetration holes in grapes can be seen, but require careful inspection of the fruit. Sometimes a few granules of frass can be found around a fresh penetration hole or a mass of frass may be found around older penetration holes (Fig.

3B). Sometimes, however, frass is not visible. The area around the penetration hole can become sunken and brown as damaged tissue decays (Fig. 3C) (Johnson, date unknown).

<u>Citrus:</u> All stages of citrus fruit are vulnerable to attack. FCM larvae are capable of developing in hard green fruit before control measures can be started. Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers. There is sometimes a scar visible on infested fruit (Stibick, 2006).

<u>Corn:</u> Larvae damage corn by entering the ear from the husk through the silk channel (Stibick, 2006).

<u>Cotton:</u> Larval penetration of cotton bolls facilitates entry of other microorganisms that can rot and destroy the boll. The cultivars Edranol, Hass and Pinkerton were the most susceptible to attack by FCM (Stibick, 2006).

<u>Macadamia:</u> Larvae damage the nuts by feeding on the developing kernel after they pierce the husk and shell. Nuts reaching 14 to 19 mm diameter size are at the most risk because nutrient content is the greatest; concurrently, false codling moth reaches the adult stage by this point and is able to oviposit on the nuts (Stibick, 2006).

<u>Avocado:</u> Moths lay eggs superficially on the fruit of avocado. Larvae hatch and develop, and can enter through the skin. Larvae are unable to develop in avocado fruit. However, their entrance creates lesions that lessen the marketability of fruit. Lesions develop into a raised crater on the fruit surface, with an inconspicuous hole in the center where the larva has entered. Granular excreta can also be seen (Stibick, 2006).

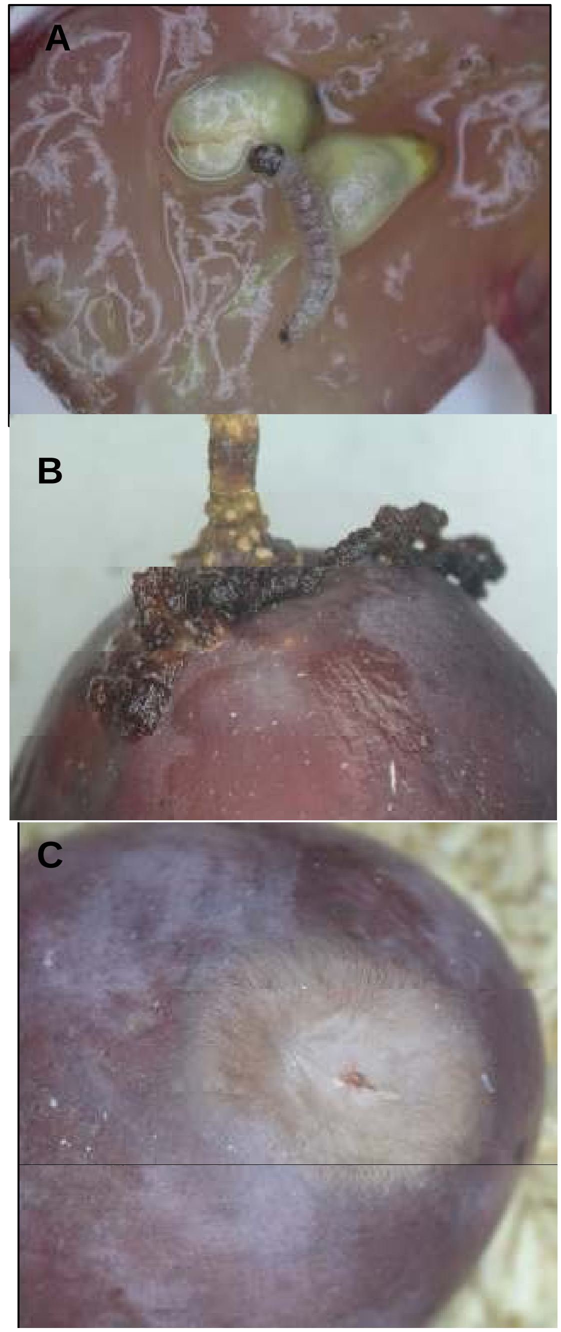

Figure 3. A) FCM larva inside grape fruit. B) A larval penetration hole near the stalk with frass exuding from it as the larva feeds inside the fruit. C) Fruit damage around the penetration hole in a grape where a FCM larva entered the fruit. Photos courtesy of Shelley Johnson, University of Stellenbosch, South Africa.

<u>Stone Fruit:</u> All stages of stone fruits are vulnerable to attack. False codling moth larvae are capable of developing in hard green fruit before control measures can be started. Once a fruit is damaged, it becomes vulnerable to fungal organisms and scavengers. Larvae damage stone fruits as they burrow into the fruit at the stem end and begin to feed around the stone. Infestations can be identified by the brown spots and dark brown frass. Peaches become susceptible to damage about six weeks before harvest. Detecting infested peaches can be difficult if the fruit is still firm and abscission has not occurred; consequently, the danger of selling potentially infested fruit poses a serious threat to the peach industry (USDA, 1984; Stibick, 2006).

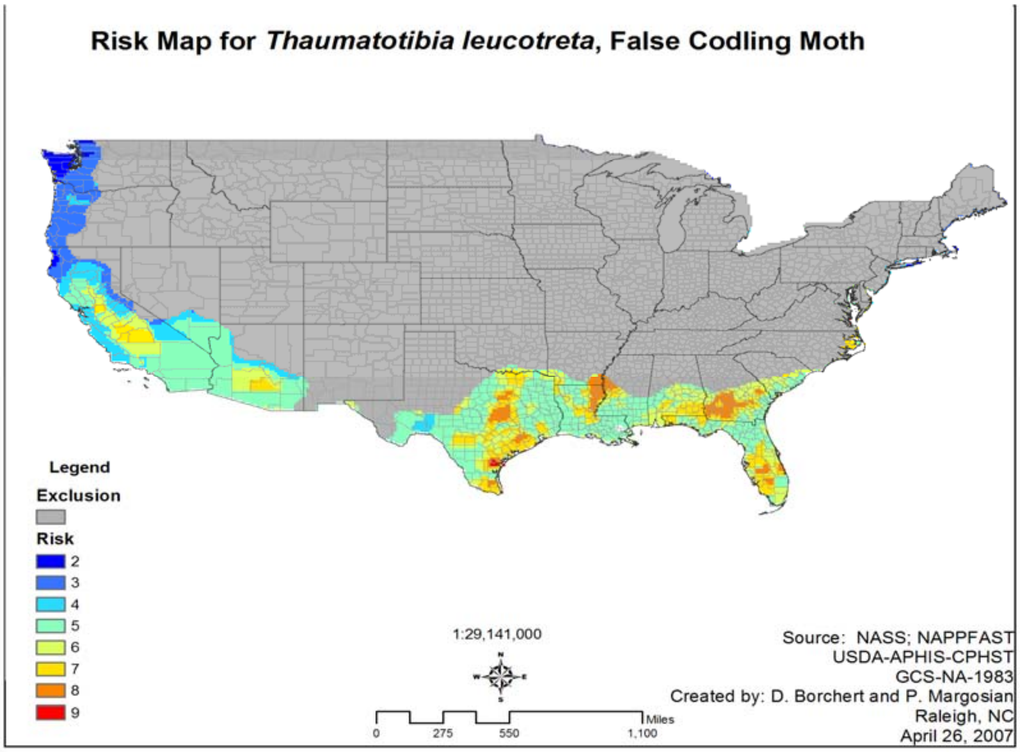

Figure 4. Risk map for *T. leucotreta* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Survey

For early detection surveys in grape, vineyards in close proximity to high risk areas such as citrus and stone fruit should be monitored utilizing pheromone traps. The pheromone traps should be placed a frequency of 1 trap per 4 hectares and traps should be no closer than 150 to 200 m to each other (Johnson, date unknown). Traps should be inspected weekly. Grapes should also be inspected visually for the presence of FCM during the growing season. The first four rows bordering citrus or stone fruit orchards should be examined carefully.

A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 4) shows that portions of Texas, Arkansas, Louisianna, Mississippi, Georgia, and Florida are at the greatest risk from *T. leucotreta*. In addition, portions of California, Arizona, and Alabama have a risk level of 7 or higher.

Preferred method: (From Venette et al., 2003)
Male *T. leucotreta* are attracted to a two component blend of (E)-8-dodecenyl acetate and (Z)-8-dodecenyl acetate. These components are most effective when used in a ratio between 70:30 and 30:70 (E:Z). More recently, Newton et al. (1993) refined the sex pheromone and reported that a 90:10 ratio was optimal. Stibick (2006) recommends utilizing a 50:50 ratio. **The lure is available from the CPHST- Otis lab.**

A loading rate between 0.5 and 1.0 mg per septum was found to attract the greatest number of males. The pheromone blend (1 mg applied to a rubber septum) has been used effectively with Pherocon 1C traps to capture male *T. leucotreta* (Newton et al., 1993). Delta traps have also been used, but these have performed less well than either the Hoechst Biotrap or Pherocon 1C traps. Traps using closed polyethylene vials to dispense pheromones captured more moths than traps using rubber septa (using a 50:50 blend of (E)- and (Z)-8-dodecenyl acetate). Lures should be replaced every 8 weeks. Traps should be placed approximately 5 ft (1.5m) high. Pheromone traps (homemade design with unspecified pheromone blend) have been used to monitor the number of *T. leucotreta* adult males in citrus orchards (Daiber, 1978) and detect the presence of the pest in peach orchards (Daiber, 1981).

Pheromone lures with (E)- and (Z)-8-dodecenyl acetate may also attract *Cydia cupressana* (native), *Hyperstrotia spp*., Cydia *atlantic*a (exotic), *Cydia phaulomorpha* (exotic) and *Cryptophlebia peltastica* (exotic).

Alternative method: Visual inspections of plant materials may be used to detect eggs, larvae, and adults of *T. leucotreta* (USDA, 1984). Look for plants showing signs of poor growth or rot; holes in fruit, nuts or bolls; adults hidden in foliage; and crawling larvae. Surveys are best conducted during warm, wet weather when the population of the pest increases (USDA, 1984). Eggs will commonly be found on fruits, foliage, and occasionally on branches (USDA, 1984). However, eggs are small and laid singly, which makes them difficult to detect.

Fruit should be inspected for spots, mold, or shrunken areas with 1 mm exit holes in the center. On citrus fruits and other fleshy hosts, dissections are needed to detect larvae; larvae are likely to be found in the pulp (USDA, 1984). Infested fruits may be on or off the tree. In cotton, older larvae may be found in open bolls and cotton seed (USDA, 1984). Occasionally adults may be observed on the trunk and leaves of trees in infested orchards (USDA, 1984). For field crops, such as corn, the whole plant is the recommended sample unit. Because larvae of *T. leucotreta* have a strongly aggregated spatial distribution among corn plants, a large sample size (>60 plants) is recommended; however at low densities of the pest (<1 larva/plant) sample sizes may be prohibitively large to detect the pest.

Soil Sampling: Collect soil samples within 200 yards of any larval or egg detection and at any spot where dropped, especially prematurely dropped, fruit occur. Soil samples should consist of loose surface soil and any debris. Examine soil for larvae, cocoons and pupae.

Not recommended: Robinson black light traps are ineffective at attracting adult *T. leucotreta* (Begemann and Schoeman, 1999). Therefore, black light traps should not be used. The effectiveness of black light traps may be improved if used in conjunction with pheromone lures (Möhr, 1973). Mohr (1973) speculates that pheromone may provide a long-distant attractant, but that attraction to black light becomes much stronger when moths are in close proximity to light traps.

Key Diagnostics

Thaumatotibia leucotreta can be confused with many *Cydia* spp. including *C. pomonella* (codling moth) because of similar appearance and damage, however, unlike codling moth its host range does not include apples, pears or quince (USDA, 1984). In West Africa, *T. leucotreta* is often found in conjunction with *Mussidia nigrevenella* (pyralid moth), however, they can be distinguished by close examination of morphological characters (CABI, 2004). In South Africa, there is also an overlapping host range for *T. leucotreta, T. batrachopa* (macadamia nut borer) and *Cryptophelbia peltastica* (litchi moth), particularly on litchi and macadamia (Venette et al., 2003; Stibick, 2006). The male litchi moth can be distinguished from similar species by a subtriangular or Y-shaped T8 with a pair of tufts of filiform scales from membranous pockets on each side (Stibick, 2006).

Male *T. leucotreta* can be distinguished from all other species by its specialized hindwing, which is slightly reduced and has a circular pocket of fine hairlike black scales overlaid with broad weakly shining whitish scales in anal angle, and its heavily tufted hind tibia (Bradly et al., 1979).

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Tertiary Pests of Grape (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to grape and exotic to the United States

Brevipalpus chilensis

Common name: grape flat mite

Reason for Inclusion in Manual

National Threat

Figure 1. The grape flat mite, *Brevipalpus chilensis*. Photo courtesy of http://www.mipcitricos.cl/aca3.htm.

Conogethes punctiferalis

Common name: yellow peach moth

Reason for Inclusion in Manual

National Threat

Figure 1. Adult *Conogethes punctiferalis.* Photo courtesy of K. Nakao (www.jpmoth.org).

Figure 2. Larva of *Conogethes punctiferalis.* Photo courtesy of http://aoki2.si.gunma-u.ac.jp/youtyuu/HTMLs/momonogomadaranomeiga.html

Cryptoblabes gnidiella

Common name: citrus pyralid

Reason for Inclusion in Manual

National Threat

Figure 1. Adult *Cryptoblabes gnidiella.* Photo courtesy of http://www2.nrm.se/en/svenska_fjarilar/c/images/cryptoblabes_gnidiella_female.gif.

Pests of Grape Requested by the CAPS Community

Homalodisca coagulata

Scientific Name
Homalodisca coagulata (Say)

Synonyms
Homalodisca triquetra, *Phera vitripennis*, *Phera coagulata*, *Tettigonia coagulata*

Common Names
Glassy-winged Sharpshooter

Type of Pest
Leafhopper

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

Reason for inclusion in manual
Request of the CAPS Community, Potential vector for *Xylella fastidiosa*

Pest Description
Eggs: Females lay their "sausage-shaped" eggs in masses of about 10 to 15 in the epidermis of the lower surface of young, fully developed leaves. The glassy-winged sharpshooter (GWSS) eggs measure approximately 2.5 mm long and 0.53 mm wide (Boyd and Hoddle, 2007). When it is first laid, the egg mass appears as a greenish blister on the leaf (Fig. 1). The female covers the inserted egg mass with a secretion that resembles white chalk and is more visible than the leaf blister. Shortly after the eggs hatch, the leaf tissue begins to turn brown. The dead leaf tissue remains as a permanent brown scar (Fig. 2).

Figure 1. Freshly laid "blister-like" eggs of *H. coagulata*. Photo courtesy of The University of California.

Nymphs: Nymphs are the immature insects that will later on become adults. Nymphs (Fig.

2) look similar to the adults except they are smaller, wingless, uniform greenish gray in color, and have prominent red bulging eyes. Nymphs range in size from .07 inches (2 mm) to nearly ½ inch (13mm) long. The nymphs are much more active than the adults and are more likely to jump away when approached.

Adults: Adults (Fig. 3) are about 1.5 to 2.0 cm in length and are relatively large for the sharpshooter family. They are generally dark brown to black when viewed from the top or side. The abdomen is whitish or yellow. The head is brown to black and covered with numerous white to yellowish spots (Nielson, 1968). The wings of the sharpshooter are translucent brown with red veins. The structure of *H. coagulata* female genetalia is described in detail by Hummel et al. (2006).

Figure 2. Older eggs darken, hatch, and appear as a brown scar (top) and GWSS nymphs (bottom). Photos courtesy of Ken Peek, Alameda County Dept. of Agriculture and the University of California

Biology and Ecology

Sanderson (1905) observed that *H. coagulata* has two to three generations per year in Texas, and the adults hibernated in 'rubbish' on the ground near food plants. Turner and Pollard (1959) using yellow sticky traps to study *H. coagulata* in Georgia, found that the insect had two full generations per year, followed by a partial third generation. Bivoltine patters of *H. coagulata* occur in Florida (Alderz and Hopkins, 1979) and southern California (Blua et al., 1999).

Adults are present and must feed throughout the year. In California, egg-laying activities are either absent or reduced to very low levels during the winter months of December, January, and February. During this same period, the number of overwintering adults also decreases. Mating occurs in the spring and summer. Egg-laying resumes in late February and continues through May. The first generation completes development from late May to late August. Adults from this generation lay egg masses from mid-June through late September, which give rise to overwintering adults (CDFA, 2006).

Symptoms/Signs

Even though the GWSS is large enough to be seen with the naked eye, it is very inconspicuous in nature. The brown coloration of the insect blends very well with the color of the twigs where it is usually found, and it hides by moving to the other side of the twig or branch when it detects movement or is otherwise approached or disturbed.

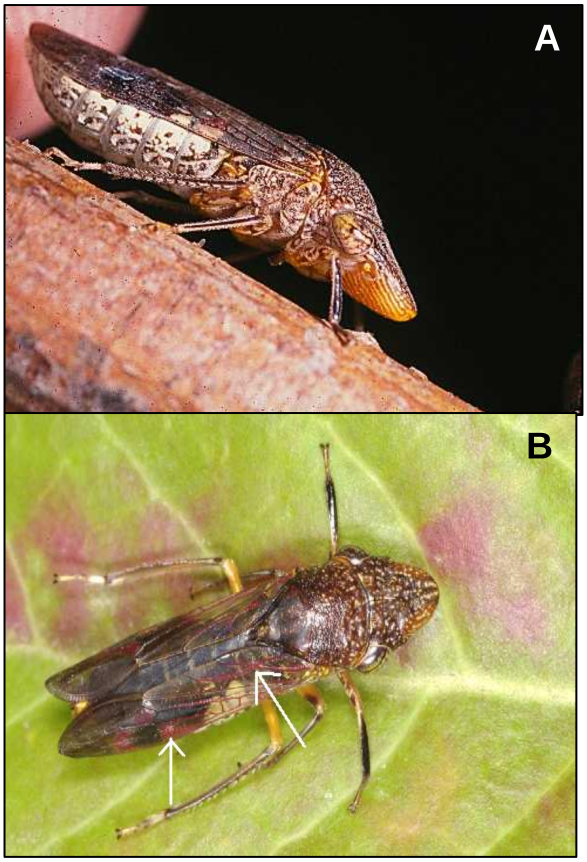

Figure 3. Adults of *H. coagulata.* The profile of the sharpshooter (A) and the characteristic red veins in the wings. (B). Photos courtesy of CDFA.

GWSS eggs are laid together on the underside of leaves, usually in groups of 10 to 12. The egg masses appear as small, greenish blisters. The eggs are covered with a white material scraped from deposits on the female forewings. This white powder is termed brochosomes, consisting of intricately structured hydrophobic particles. These blisters are easier to observe after the egs hatch, when they appear as tan to brown scars on the leaves.

When a large number of sharpshooters are in a tree they constantly draw fluids out of the branches and excrete them out the other end giving the appearance of “rain” coming off the tree. The excreta also can cause a whitewashed appearance on leaves, fruit and even on the sidewalk under it. Leaves or fruit coated with a whitish, powdery material may indicate that there has been heavy GWSS feeding on that plant (Fig. 4).There are other ways to create this whitewashed appearance, mineral deposits from water is one common way. White spots on plants are not always a sign that sharpshooters are present, but if you see it, closer inspection may reveal the sharpshooter hiding on the stems or branches of the plant.

Figure 4. Whitewashed appearance to fruit due to a GWSS infestation. Photo courtesy of The University of California.

Because the sharpshooter can remove a large amount of water from the xylem tissue, during drought conditions plants may become weakened if the water is not replenished.

Pest Importance

The GWSS is a polyphagous pest native to the southeastern region of the United States. It is usually not a serious pest in the area of its native distribution. It was first reported in California in 1989 and has since spread throughout southern California. GWSS feeds on the plant's xylem fluid and can acquire and transmit the *Xylella fastidiosa* bacterium (Fournier et al., 2006). Strains of this xylem-limited bacterial pathogen are responsible for several plant disease including Pierce's disease, almond leaf scorch, oleander leaf scorch, citrus variegated chlorosis, phony peach disease, oak leaf scorch, and etc. The bacterium replicates in the mouthparts of the insect (Hill and Purcell, 1995), and the pathogen is lost during the molting process (Purcell and Finlay, 1979). However, if an insect acquires *X. fasitidiosa* as an adult, it can retain the pathogen for the remainder of its life (Severin, 1949). In microscopic studies of *X. fastidiosa*-infected glassy-winged sharpshooters, bacteria were observed in the cibarium and apodemal groove of the diaphragm (Brlansky et al., 1982, 1983). These data suggest a noncirculative mechanism of transmission (Purcell and Finlay, 1979)**.**

The GWSS has an extremely broad host range of over 200 species (CDFA, 2001), and a single individual will feed on a variety of plant species during its lifetime. The high mobility (distances greater than 90 m) of this insect (Blackmer et al., 2004), and its use of a large number of host plant species, provides this vector with ample exposure to multiple *X. fastidiosa* strains during its lifetime (Costa et al., 2006)**.** Almeida et al (2005) showed that *H. coagulata* can acquire *X. fastidiosa* from and inoculate the bacterium into dormant grape plant. Transmission to dormant plants during the winter is a potential problem in vineyards adjacent to citrus groves or other habitats with large overwintering populations of *H. coagulata*.

In Texas, the single greatest threat to the production of susceptible grape cultivars is Pierce's Disease. The disease has caused millions of dollars in losses to the state's wine industry since 1990 and the problem has escalated in the past five years. Also within the last few years, the GWSS has established itself in southern California where it also poses a potentially serious threat to the entire wine and table grape industry in that region of the country. *H. coagulata* currently is considered to be the most significant insect pest threatening the California grape industry (Purcell and Saunders, 1999).

Known Hosts

Over 200 host plants in 35 families are known to serve as hosts for *H. coagulata* (CDFA, 1991). Major agricultural, ornamental, and weed hosts are listed below. For a complete listing see: http://pi.cdfa.ca.gov/pqm/manual/htm/454.htm#a

Hosts: *Amaranthus* spp**.** (pigweed), *Acacia* spp. (wattles), *Ambrosia* spp. (ragweed), *Anelmoschus esculentus* (okra), *Canna* spp. (canna), *Capsicum* spp. (chile pepper), *Cersis* spp. (red bud), *Chenopodium* spp. (lambsquarter), *Chamaedorea* spp (palms), *Chrysanthemum* spp. (chrysanthemum), *Citrus* spp. (citrus), *Coleus* spp. (coleus), *Crassula* spp. (crassula), *Dianthus* spp. (dianthus), *Elaeagnus* spp. (elaeagnus), *Erigeron* spp. (fleabane), *Eucalyptus* spp. (eucalyptus), *Euonymous* spp. (euonymus), *Ficus* spp. (fig), *Fraxinus* spp. (ash), *Gladiolus* spp. (gladiolus), *Hedera* spp. (ivy), *Helianthus* spp. (sunflower), *Hibiscus* spp. (hibiscus), *Ilex* spp. (holly), *Ipomoea* spp. (morning glory), *Jasminum* spp. (jasmine), *Juniperus* spp. (juniper), *Lactuca* spp. (lettuce), *Lagerstroemia* spp. (crape myrtle), *Lantana* spp. (shrub verbena), *Laurus nobilis* (sweet bay), *Lingustrum* spp. (privet), *Liriope* spp. (giant turf lily), *Lonicera* spp. (honeysuckle), *Malus* spp. (apple), *Morus* spp. (mulberry), *Nephrolepsis* spp, (sword fern), *Nerium* spp. (oleander), *Olea* spp. (olive), *Persea* spp. (avocado), *Philodendron* spp. (philodendron), *Phlox* spp. (phlox), *Phoenix* spp. (date palm), *Photinia* spp. (photinia), *Pinus* spp. (pine), *Pittosporium* spp. (pittosporium), *Prunus* spp. (peach, apricot, cherry), *Quercus* spp. (oak), *Rhus* spp. (sumac), *Rosa* spp. (rose), *Rubus spp.* (blackberry), *Salix* spp. (willow), *Schinus* spp. (pepper tree), *Solanum* spp. (potato, eggplant), *Solidago* spp. (goldenrod), *Sonchus* spp. (sonchus), *Strelitzia* spp. (bird of paradise), *Thuja* spp. (arborvitae), *Tradescantia* spp. (spiderwort), *Ulmus* spp. (elm), *Viburnum* spp. (viburnum), *Vinca* spp. (periwinkle), *Vitis vinifera* (grape), *Washingtonia* spp. (Washington palm), *Wisteria* spp. (wisteria), *Yucca* spp. (yucca), *Xylosma* spp. (xylosma), and *Zea* spp. (corn).

Known Vectors (or associated insects)

The GWSS is a known vector of several strains of *Xylella fastidiosa* (see pest importance section),

Known Distribution

Turner and Pollard (1959) describe the range of *H. coagulata* as a strictly southern species native to the United States, abundant from the latitude of Augusta, Georgia to Leesburg, Florida, having a western boundary of Val Verde and Edwards counties in Texas. Current distribution data shows that the sharpshooter is present to some extent in northern Mexico and south Florida as well. The described native range includes Flordia, Georgia, North Carolina, South Carolina, Mississippi, Alabama, Texas, Missouri, and Arkansas.

Sorensen and Gill (1996) noted that the range of *H. coagulata* has extended to include several counties in southern California, most likely having been introduced to the state through the nursery industry. The infested counties in California include: Los Angeles, Orange, Riverside, San Bernadino, San Diego, and Ventura Counties. Limited infestations of GWSS also occur in areas of Fresno, Kern, Imperial, Sacramento, Santa Barbara, Santa Clara, Solano, and Tulare Counties (CDFA, 2006). In May 2004, the GWSS was identified from Hawaii (Hawaii Department of Agriculture, 2004).

North America: Mexico and the United States. **Oceania:** French Polynesia (Tahiti), Easter Island (CABI, 2004; Logarzo et al., 2006).

Potential Distribution within the United States

GWSS is abundant in the southeastern United States and infestations occur in California and Hawaii.

Survey

Preferred Method. Surveys in California are currently being conducted using a combination of sticky traps and visual surveys (CDFA, 2006).

Note: Sticky cards will not detect sharpshooters in the juvenile or egg stages. Thus, utilizing both sticky traps and visual survey is the best way to find new infestations.

Trapping: Yellow sticky traps are commonly used for surveillance and detection of adults in orchards, although *H. coagulata* only shows slight, if any, preference for yellow compared to other colors (CABI, 2004). Panels measuring a minimum of 5" x 9" are the trap of choice for GWSS. The flight temperature threshold for GWSS is approximately 18 °C (65 °F). Trapping will not be effective during periods when temperatures are lower than this threshold. Trapping season begins no earlier than March 1 and ends October 31, depending on local conditions. Trap densities from 1-5 traps per sq. mile are used for areas with 25-501 residences per square mile (CDFA, 2006). Rural areas with less than 25 homes per square mile should not be trapped. The number of traps should be doubled in the area within ¼ mile of high-risk nurseries (those which receive plant material from GWSS-infested areas). Irrigated areas with a diversity of plants which include multiple preferred hosts should be selected whenever possible. Traps should be placed near lights when possible. Preferred hosts should always be selected for trap deployment. Good hosts include the following:

- Spring: Crape myrtle, citrus, privet, photinia, grape, mulberry, xylosma, oleander, pittosporium, and euonymus.

- Summer: Crape myrtle, citrus, privet, photinia, grape, mulberry, xylosma, oleander, ornamental plum, pear, peppertree, red bud, pittosporium, and euonymus.

- Fall: Citrus, crap myrtle, oleander, olive, red bud, photinia, privet, and xylosma.

Traps should be placed primarily in the outer canopy of host trees (*e.g.,* citrus) and highly visible. Traps should be serviced every two weeks and replaced/relocated every six weeks to another host at least 500 feet away. For grape, traps should be placed from the edge to within 30 feet of vineyards for early detection (University of California and USDA, date unknown). Traps may be placed on trellises and above the canopy or poles/stakes can be used to suspend the yellow panel traps just above the grape canopy. Deployment in perimeter rows or along heavily traveled routes should be avoided. The traps should be hung perpendicular to the vine so that both sides of the trap are visible (Yolo County, 2004). Traps should be moved as vine growth and development occurs to prevent traps from being obscured by foliage. One sticky trap per 10 acres is recommended but placement in the area is dependent upon terrain and surrounding GWSS host plants (particular attention should be given to areas bordered by citrus). Additionally if a winery is present at the vineyard location, place traps at the edge adjacent to the winery or landscape plantings. For grapes, replacement of traps every two weeks is recommended.

Visual surveys: Visual surveys can be conducted by examining the terminal leaves, petioles, and stem in citrus and grapes. Surveys should begin no earlier than June 1 and end no later than October 31. GWSS infestations can also be determined by examining the underside of plant leaves or recently matured foliage (older foliage should be avoided) for the presence of adults, nymphs, nymphal cast skins, egg masses, and egg scars. Inspections for egg masses and nymphs are best restricted to known hosts. Old egg scars are the easiest to detect since egg deposition sites are visible on both leaf surfaces. Newly laid eggs, in contrast, are only visible on the underside of the leaves. Consequently, a representative sample of leaves should be turned over and examined with for egg masses. Backlighting against a sunny sky will also aid in finding egg masses (CDFA, 2006).

When searching for active life stages stages (nymphs and adults) search the plant stems. On trees, the GWSS usually selects shoots that are growing upward (vertically oriented as opposed to horizontal twigs. New flush growth and southern exposures are also preferred (CDFA, 2006). Adults are the easiest life stage to detect, because they are highly visible when flying around or between host plants. Flight activity is most pronounced during the late morning and afternoon hours.

Insect nets: Visual searches of the host plants can be enhanced by using insect nets (aerial and sweep and beat sheets). The effectiveness of these devices is largely dependent on the type and density of GWSS life stages present. At low densities and during cooler times, nets and beat sheets may be used to agitate foliage causing adults to take flight.

On warm nights, *H. coagulata* is attracted to black and incandescent lights (CABI, 2004).

Detailed survey methods for GWSS are given at http://134.186.235.120/pdcp/Documents/Survey-Delimitation%20Protocols%2007-08.pdf. Refer to this document for additional clarification and detail.

Key Diagnostics

The morphology of the adult has been used primarily to differentiate species of *Homalodisca* and other sharpshooters. If an egg mass is discovered, considerable time and cost were required to rear the insects to the adult stage for morphological identification.

Predator gut analysis studies are often conducted to assess the efficacy of a potential biological control agent. Fournier et al. (2006) developed an ELISA technique to detect the egg stages of *H. coagulata* and *H. liturata* (the smoke tree sharpshooter) and to a lesser extent the adult stage of gravid females in the gut of predators. De Leon et al. (2006) used sequence characterized amplified region (SCAR) makers to develop markers that are *H. coagulata* and *H. coagulata*/*H. liturata* specific for use in predator gut content examinations. The authors stated, however, that these markers would be useful in identifying any life stage of *H. coagulata* and/or *H. liturata*.

Smith (2005) showed that the amino acid sequences of *H. liturata* and *H. coagulata* had only a single substitution that was unique (75^{th} amino acid of alignment = serine in *H. liturata* and asparagine in *H. coagulata*). Though the majority of the genetic variation between the two species was neutral, the author felt that it could still be useful to develop a molecular diagnostic tool to discriminate between nymphs of *H. liturata* and *H. coagulata*.

Additionally, Costa et al. (2006) describe a PCR-based method to detect and differentiate strains (Pierce's disease and oleander leaf scorch) of *X. fastidiosa* present in individual GWSS adults.

Easily Confused Pests

Adults *H. coagulata* appear dark brown to black in their overall appearance. The abdomen ranges in color from white to yellow. The head is brown to black and covered with numerous ivory to yellowish spots. These spots help distinguish glassy-winged sharpshooter from a close relative, the smoke tree sharpshooter *(H. liturata,* also listed as *H. lacerata)*, which is native to the desert region of southern California and has pale, wavy lines instead of spots (Gill, 1995; CABI, 2004). The smoke tree sharpshooter is also slightly smaller (Gill, 1995). Immature stages are very difficult to distinguish (Smith, 2005).

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Mollusks

Primary Pests of Grape (Full Pest Datasheet)

None at this time

Secondary Pests of Grape (Truncated Pest Datasheet)

Achatina fulica

Scientific Name
Achatina fulica Bowdich

Synonyms:
Lissachatina fulica

Common Names
Giant African snail, African giant snail, Kalutara snail

Type of Pest
Mollusk

Taxonomic Position
Class: Gastropoda, **Order:** Pulmonata, **Family:** Achatinidae

Reason for inclusion in manual
CAPS Target: AHP Prioritized Pest List

Pest Description
Achatina fulica is distinctive in appearance and is readily identified by its large size and relatively long, narrow, conical shell (Fig. 1, 2).

Figure 1. The large size of the giant African snail. Photo courtesy of USDA-APHIS.

Eggs: Elyptical, about 4 mm by 5 mm in diameter, usually pale yellow, laid in clutches of 100-400 (Fig. 3) (USDA, 1982).

Juveniles: Similar to adults, but have a thinner, translucent shell, which is more brittle. Upon emergence, the juvenile shell is approximately 4 mm long

(Denmark and Poucher, 1969; USDA-APHIS, 2005). Increase at a rate of 10 mm per month for first four months. The coloration is similar to adults. The columella is truncated.

<u>Adults:</u> Columella abruptly truncate (Burch, 1960). Columella and the parietal callus are white or bluish-white with no trace of pink (Bequaert, 1950). Shell size may be up to 8 inches (203 mm) in length and almost 5 inches (127 mm) in maximum diameter (Bequaert, 1950). Shell has seven to nine whorls and rarely as many as ten whorls (Bequaert, 1950). Shell color is reddish-brown with light yellowish, vertical (axial) streaks; or, light coffee colored. Protoconch is not bulbous. Body coloration can be either mottled brown or more rarely a pale cream color. The truncated columella is evident throughout the lifespan of the snail. The columella is generally concave. Snails with a lesser concaved columella tend to be somewhat twisted. Snails with a broader shell tend to have a more concave columella (Bequaert, 1950). In calcium-rich areas, the shells of the adults tend to be thicker and opaque (USDA-APHIS, 2005).

Figure 2. Adult giant African snail. Photo courtesy of Matt Ciomperlik, USDA APHIS PPQ

The giant African snail, *Achatina fulica*, is a polyphagous pest. This species is one of the most serious land snail pests known, reported to consume all growth stages of vegetables, cover crops, garden flowers, herbaceous ornamentals, and damaging many fruit and ornamental trees (USDA, 1982). Its preferred food is decayed vegetation and animal matter, lichens, algae and fungi. The bark of relatively large trees such as citrus, papaya, rubber and cocoa is also subject to attack. Poaceous crops (sugarcane, maize, rice) suffer little or no damage from this species. There are reports of *A. fulica* feeding more than 500 species of plants (CABI, 2004).

Figure 3. Giant African snail eggs. Courtesy of USDA APHIS.

A large infestation presents a nuisance problem with slime trails, excretions, and odors of decay when they die in large numbers. *A. fulica* has been shown to be a health hazard by transmitting the rat lungworm, *Angiostrongylus cantonensis*, which causes eosinophicillic meningoencepahlitis in humans. *A. fulica* has also been implicated in transmitting the following plant pathogens: *Phytophthora palmivora* on commercial pepper, coconut, betel pepper, papaya, and vanda orchid; *Phytophthora colocasiae* on taro; and *Phytophthora parasitica* on eggplant and tangerine (USDA, 1982).

A. fulica, believed to be originally from East Africa, has become established throughout the Indo-Pacific Basin, including the Hawaiian Islands. This mollusk has also been introduced to the Caribbean islands of Martinique and Guadeloupe. Recently, the snails were detected on Saint Lucia and Barbados. Although many introductions are accidental via cargo or ships, some introductions were purposeful. The market for this snail species as food is expanding. In Africa and Asia, the medicinal properties of these snails are also being investigated. The U.S. Department of Agriculture has recently discovered and confiscated illegal giant African land snails from commercial pet stores, schools and one private breeder. These snails were being used for science lessons in schools by teachers who were unaware of the risks associated with the snails and the illegality of possessing them. In 1966, a Miami boy smuggled three giant African land snails into the country. His grandmother eventually released them into a garden, and in seven years, there were more than 18,000 of them. The Florida state eradication effort took 10 years at a cost of $1 million.

Symptoms/Signs

Information specific to grape is not available. *A. fulica* is easily seen due to its large size, and attacked plants exhibit extensive rasping and defoliation. The weight of the number of snails on a plant can break the stems of some host species. *A. fulica* can also be detected by signs of ribbon-like excrement, and slime trails on plants and buildings.

In garden plants and ornamentals of a number of varieties, and vegetables, all stages of development are eaten. Cuttings and seedlings are the preferred food items. Young snails up to about 4 months feed almost exclusively on young shoots and succulent leaves. The bark of relatively large trees such as citrus, papaya, rubber and cocoa is also subject to attack. In these plants, damage is caused by complete consumption or removal of bark. The papaya appears to be the only fruit that is seriously damaged by *A. fulica*, largely as a result of its preference for fallen and decaying fruit (CABI, 2004).

Survey

Preferred Method: The most effective method of survey for mollusks is through visual searching methods. Traps baited have been used in the past but are not effective for tropical species, such as the achatinids. *A. fulica* is a large and conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight, or in the morning or evenings following a rain event.

A recent risk map developed by USDA-APHIS-PPQ-CPHST (Fig. 4) shows that portions of Texas, Arkansas, Louisiana, Mississippi, Alabama, Georgia, Florida, South Carolina, North Carolina, and Virginia are at the greatest risk from *A. fulica*. In addition, portions of California, Arizona, and New Mexico are also at risk.

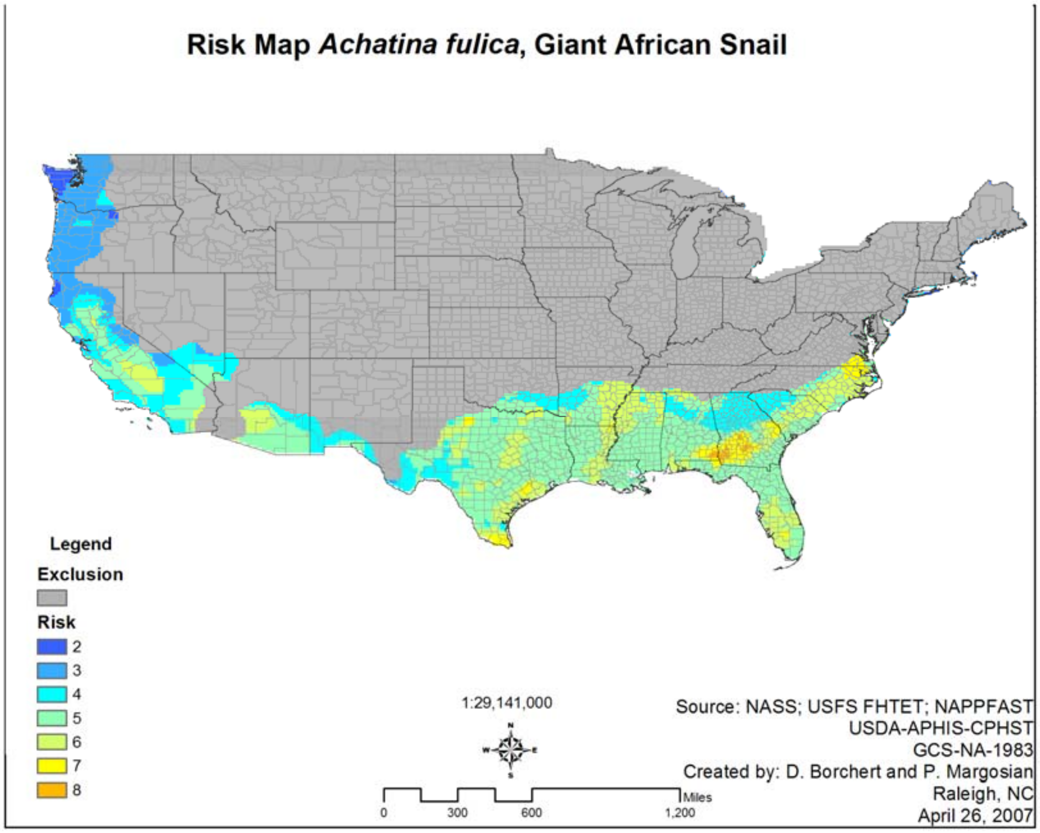

Figure 4. Risk map for *A. fulica* within the continental United States. Map courtesy of Dan Borchert, USDA-APHIS-PPQ-CPHST.

Detailed survey information is available in the New Pest Response Guidelines available at http://www.aphis.usda.gov/import_export/plants/manuals/emergency/downloads/nprg_gas.pdf.

When using visual inspection methods for detection surveys an amount of bias and variation in sampling intensity is possible. In an effort to standardize sampling efforts, and thus approach a higher level of confidence in results, the following factors should be considered:

Seasonality: Conduct detection surveys on an ongoing basis, with repeated visits at the beginning, during, and/or just after the rainy season. Keep in mind that *Achatina fulica* remains active at a range of 9-29°C (48-84 °F). *Achatina fulica* begins hibernating at 2°C (35°F), and begins aestivation at 30°C (86 °F).

<u>Time of Sampling:</u> Plan surveys for early morning and overcast days. Achatinids are active on warm nights, early mornings, and overcast and rainy days. To maintain a consistent sampling time, conduct surveys in the early morning. On overcast days, conduct additional surveys throughout.

<u>Micro Habitats:</u> During the day, find snails in the following moist micro habitats: near heavily vegetated areas; under or near rocks and boulders; under discarded wooden boards and planks, fallen trees, logs, and branches; in damp leaf litter, compost piles, and rubbish heaps; under flower pots and planters; on rock walls, cement pilings, broken concrete, or grave markers; in gardens and fields where plants have been damaged by feeding snails and slugs; and at the base of the plants, under leaves, or in the "heart" of compact plants, such as lettuce or cabbage.

<u>Evidence:</u> While conducting a survey, look for the following clues that suggest the presence of snails: chewing damage to plants, eggs, juveniles and adults, empty snail shells, mucus and slime trails, large, ribbon-like feces, and an increase in rat population densities in an area.

<u>Alternative method:</u> Use traps to supplement a visual inspection, if time and resources allow. Use commercial brands of slug bait to attract snails; however, due to the slow-acting effects of the molluscicide, these baits alone are not effective in trapping snails.

<u>Note:</u> 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

A. fulica is distinctive in appearance and is readily identified by its large size and relatively long, narrow, conical shell. Reaching a length of up to 20 cm the shell is more commonly in the range of 5 to 10 cm. See identification section in USDA-APHIS (2005).

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Cernuella virgata

Scientific name
Cernuella virgata Da Costa

Synonyms:
Cernuella virgatus, *Cernuella variabilis, Cernuella virgata* ssp. *variegata*, *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: AHP Prioritized Pest List

Pest Description
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). The maritime gardensnail is relatively small and is characterized by prominent spiral banding on the shell (Fig. 1).

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

Symptoms/Signs
C. 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 contaminates crops and clogs machinery. Areas previously infested with snails can prevent the re-establishment of a site as pastureland as livestock often reject slime-contaminated hay and forage (Baker, 2002).

Survey

C. virgata is a conspicuous crop pest that hides during the day. Surveys are best carried out at night using a flashlight or in the morning or evening following a rain event. It is easily seen, and attacked plants exhibit extensive rasping and defoliation. Like other mollusks, it can also be detected by signs of ribbon-like excrement and slime trails on plants and buildings.

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

Key Diagnostics

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

C. virgata closely resembles the white Italian snail (*Theba pisana*) in appearance and pest status. *C. virgata* can be differentiated from *T. pisana* by more pronounced spiral banding. Also, 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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Tertiary Pests of Grape (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to grape and exotic to the United States

None at this time

Nematodes

Primary Pests of Grape (Full Pest Datasheet)

None at this time

Secondary Pests of Grape (Truncated Pest Datasheet)

Meloidogyne mali

Scientific name

Meloidogyne mali Itoh, Ohshima, and Ichinohe

Synonyms:
None

Common Name(s)

Apple root-knot nematode

Type of Pest

Nematode

Taxonomic Position

Class: Nematoda, **Order:** Tylenchida, **Family:** Meloidogynidae

Reason for Inclusion in Manual

CAPS Target: AHP Prioritized Pest List

Pest Description

Measurements in µm from Itoh et al. (1969). Measurements in parentheses indicate the range of measurements observed in the character.

Females: Stylet length 15 (13-17), dorsal gland opening 5.5 (4-7) behind stylet base, excretory port 23 (20-29) annules from anterior. Perineal pattern oval, striae smooth, finely spaced, dorsal arch low, flat, some pronounced transverse striae toward both ends of vulva, ventral arch flat, tail terminus forming circular striae. Phasmids large, "a distinct striae always located bending downwards spanning phasmids." Lateral fields with single or double incisures.

Males: Length 1447 (1270-1630), a=38 (31-44), c=40 (32-58), stylet length 20 (18-22), dorsal gland opening 8 (6-13) behind stylet base, excretory pore 20 (7-26) annules behind median bulb and 1 or 2 annules posterior to hemizonid, spicules 32 (28-35), gubernaculums 8.5 (7-10). Lateral field with 4 incisures areolated on tail portion.

Second-stage juveniles: Length 418 (390-450), a = 28.5 (27-31), c=13.3 (12-15), c' = 3.7 (3-5), stylet length 14, dorsal gland opening 4.7 (4-6) behind stylet base. Tail length 31 (30-34), conoid, teriminus irregular, rounded, unstriated. Rectum not swollen. Lateral field with 4 incisures, outer bands wider than inner band.

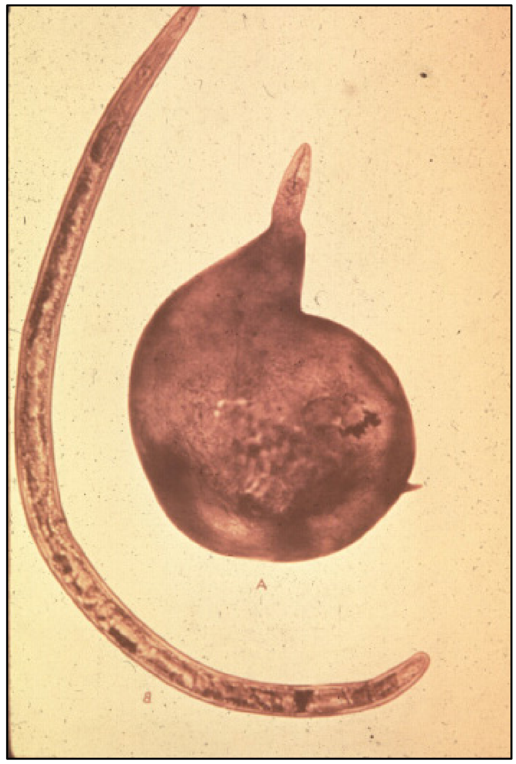

Figure 1. Male (left) and female (right) root knot nematode. The male nematode remains mobile after the final molt, whereas the female stays in place (sedentary), establishes a feeding site, matures, and lays eggs. Photo courtesy of North Carolina State University.

Symptoms/Signs

Above ground, new primary shoot growth decreases in number and length. Annual gain in plant height, trunk thickness, and numbers of leaves are reduced. Secondary shoots increase in numbers and length. The above ground symptoms are not specific for *M. mali* and may be caused by other nematodes or organisms damaging the roots.

Below ground, *M. mali* produces the characteristic root-knot nematode galls on roots of host plants (Fig. 2). All stages are found associated with the root galls. Eggs, infective second-stage juveniles, and males can also be found in the soil.

Survey

Soil samples must be collected and analyzed by a taxonomic expert. In apple orchards, *M. mali* is most abundant in the top 25 cm of soil. A few nematodes occur 50 cm deep. Horizontally, the nematodes are most abundant in two zones, 20-40 cm and 120-160 cm around the tree. These patterns of nematode distribution are thought to be related to the development and distribution of apple roots in the soil (USDA, 1987).

Key Diagnostics

Males, females with head attached, and second-stage juveniles are needed for species determination. Males and juveniles should be relaxed by gentle heat before fixation in a formalin solution. Females can be fixed without heating. If galled roots are submitted for determination, males and second-stage juveniles should first be extracted by placing the roots in a moist atmosphere for several days. Extracted nematodes can then be relaxed and fixed.

Figure 2. Characteristic root knot galls caused by *Meloidogyne* spp. Photo courtesy of AVRDC- The World Vegetable Center.

Female *M. mali* have an oval cuticular perineal pattern with a flat dorsal arch as with *M. arenaria.* The species can be separated from *M. arenaria* by a characteristic double ridge of striae, which are present in the inner part of the lateral field.

A PCR-RFLP technique has been developed to separate Japanese species of the genus *Meloidogyne* from Japan (including *M. mali)* using individual second-stage juveniles (Orui, 1998). Ten *Meloidogyne* species were best discriminated using either *Mse*I and either *Ssp*I or *Vsp*I+*Hin*fI-digested patterns of the PCR product.

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Orui, Y. 1998. Identification of Japanese species of the genus *Meloidogyne* (Nematoda: Meloidogynidae) by PCR-RFLP analysis.

USDA. 1987. Pests not known to occur in the United States or of limited distribution. No. 83: Apple Root-Knot Nematode.

Tertiary Pests of Grape (Name and Photo only) – on other lists, not a CAPS priority but a potential threat to grape and exotic to the US

Xiphinema italiae

Common name: dagger nematode

Reason for Inclusion in Manual

National Threat

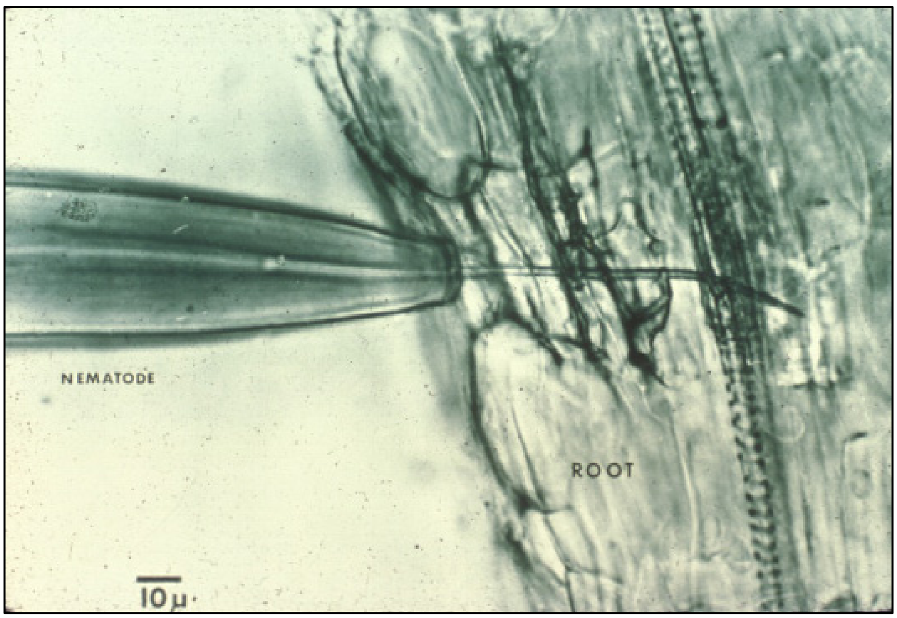

Figure 1. Stylet of *Xiphinema* spp. penetrating a root. *Xiphinema* spp. have stylets that are large enough to transmit disease.

Plant Pathogens

Primary Pests of Grape (Full Pest Datasheet)

Candidatus Phytoplasma australiense

Scientific Name

Common Name(s)
Australian grapevine yellows, Australian lucerne yellows, coprosoma lethal disease, cordyline sudden decline, liquidamber yellows, Nivun Haamir die back, papaya dieback, paulownia yellows, phormium yellow leaf, pumpkin yellow leaf curl, strawberry green petal, strawberry lethal yellows, yellow leaf disease

Type of Pest
Phytoplasma

Taxonomic Position
Class: Mollicutes, **Order:** Acheloplasmatales, **Family:** Acheloplasmataceae

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

Pest Description
Mollicutes are prokaryotes that have small genomes (530 kbp to 1350 bp), lack a cell wall, are pleomorphic, and have low a G + C content (23-29 mol%). Phytoplasmas belong to the class Mollicutes and are the proposed causative agents of diseases in several hundred plant species (McCoy et al., 1989). Phytoplasmas reside in the phloem tissue of the infected plant host and are transmitted by insect vectors, principally leafhoppers and planthoppers (White et al., 1998). Although phytoplasmas have been detected in affected plant tissues and insects with the use of technologies based on the transmission electron microscope, antibodies, and nucleic acids, they are unable to be cultured *in vitro*. Phytoplasmas cannot be morphologically or ultrastructurally distinguished from one another using either electron or light microscopy (CABI, 2004). *Candidatus* in scientific classification is a formal word that is placed before the genus and species name of bacteria that cannot be maintained in a Bacteriology Culture Collection. *Candidatus* status may be used when a species or genus is well characterized but unculturable.

Candidatus Phytoplasma australiense (herein abbreviated as *Ca* P. australiense) is the phytoplasma associated with Australian grapevine yellows (AGY), papaya dieback (PD), and phormium yellow leaf diseases (PYL) in New Zealand and Australia (Davis et al., 1997). Molecular studies have shown that the phytoplasmas associated with AGY, PD, and PYL belong to the same species (Liefting et al., 1998). This phytoplasma is distinct from German grapevine yellows and stolbur. PYL is a lethal disease of New Zealand flax (*Phormium tenax*) and mountain flax (*P. cookianum*), which is only present in New Zealand (first found in 1908). It is transmitted by a planthopper, *Oliarus atkinsoni*. PD can be a devastating disease in Queensland, Australia (first found in 1922). Australian grapevine yellows was first reported in Australia in 1975. No vectors have been identified for the latter two diseases.

Taxonomists classify phytoplasmas by directly comparing their 16S ribosomal ribonucleic acid (rRNA) sequences plus the 16S-23S rRNA intergenic spacer region (White et al., 1998). They group isolates that are $\geq$ 97.5 in the same *Candidatus* species, unless significant biological or genetic properties suggest that they should be classified separately (Andersen et al., 2006). Andersen et al. (2006) reveal three distinct subgroups within *Ca.* P. australiense by sequencing the *tuf* gene, which encodes the elongation factor Tu.

Biology and Ecology

The biology of *Ca.* P. australiense is not completely understood. Like other phytoplasmas, it is an obligate intercellular parasite that occurs in the phloem sieve tubes of infected plants and the salivary glands of insect vectors (CABI, 2004). *Ca.* P. australiense cells, like other phytoplasmas, are surrounded by a single-unit membrane, lack a rigid cell wall, and are pleomorphic in shape. When observed by transmission electron microscopy, they appear as rounded to filamentous, pleomorphic bodies with a mean diameter of 200-800 nm (IRPCM, 2004). They are sensitive to antibiotics of the tetracycline group but not to penicillin.

The cixiic planthopper, *Oliarus atkinsoni*, which occurs in New Zealand, is the only known vector of phorium yellow leaf. Researchers suspect that there are other, presumably polyphagous vectors (NPAG, 2007). Suspects include native New Zealand

Figure 1. Symptoms associated with *Candidatus* Phytoplasma australiense in grape. Mild, irregular chlorosis; leaves with backward curling, overlapping of leaves, tip death (left); necrosis (center); chlorosis along the veins (right). Photos courtesy of Fiona Constable (CABI, 2004).

psyllids, the passionvine hopper, *Scolypopa australis* (Ricaniidae), and the green planthopper, *Siphanta acuta* (Flatidae) (Lucas, 2005). The bramble leafhopper, *Ribautiana tenerrima* (Cicadellidae), and the yellow pasture leafhopper, *Zygina zealandia* (Cicadellidae), are two possible vectors of the strawberry lethal yellows strain of *Ca*. P. australiense (NPAG, 2007).

Some of the diseases caused by *Ca.* P. australiense are "new" diseases or recently described (see known hosts section). For example, cordyline sudden death became a concern in the late 1980s; apparently this phytoplasma jumped to other hosts, including cabbage trees. The ability to use new hosts is an important and threatening aspect of the pathogen (NPAG, 2007).

Figure 2. Symptoms associated with *Candidatus* Phytoplasma australiense. Irregular reddening. Photos courtesy of Fiona Constable (CABI, 2004).

Symptoms/Signs

Grape: Symptoms include yellow (chlorotic) and downward curled leaves that fall prematurely; reddening may be seen in red varieties (Fig. 1,2). The chlorotic patches on affected leaves may become necrotic. Leaves of affected shoots can overlap one another. Shoots are stunted and unlignified. Abortion of flowering bunches early in the season has been observed (Constable et al., 2004). Any time from flowering, bunches may shrivel and fall (Magarey and Wachtel, 1986; CABI, 2004). Stems of affected shoots often take on a bluish hue (Constable et al., 2004). Only a few shoots on grapevine are usually affected, and inflorescence and fruit are generally only affected on symptomatic shoots. Later in the season, affected shoots tend to be green and rubbery (CABI, 2004). The symptoms associated with *Ca.* P. australiense can be influenced by the environment. Infected grapevines are less likely to show symptoms in summer than winter (CABI, 2004). Although the infected vines are likely to show symptoms year after year, the disease can go into remission and not express symptoms (CABI, 2004).

Other hosts:

- On papaya, symptoms include dieback, bending of the growing tip, bunching and chlorosis of the crown leaves, followed by necrosis of the young leaves and stem. Laticifer discoloration, particularly in the vicinity of the vascular tissue is evident. Plant death is observed within 2 to 3 weeks of first visible symptoms.
- In pumpkin, strawberry, and peach, plant growth is stunted and leaves turn yellow and curl (roll).
- On New Zealand flax, abnormal yellowing of the leaves, stunted growth, increased root death, phloem necrosis, and xylem gummosis of the rhizome vascular system are observed (CABI, 2004).
- Coprosma lethal decline causes leaf reddeing and bronzing, heavy leaf loss, dieback, and plant death (Beever et al., 2004).
- Cordyline sudden decline may cause the death of mature cabbage trees.
- In sweetgum, the crown may have patchy chlorosis, chlorotic shoots with comparatively few leaves, dieback of apical and lateral branches, small leaves showing tip necrosis, and reduced fruit production (Habili et al., 2006).
- Paulownia yellows stunts plant growth, causes leaves to 'yellow', and reduces internode length and leaf size (Bayliss et al., 2005).
- In red clover, diminished leaf size, pallor, rugosity, leaf deformation shoot proliferation, and severe stunting were observed (Saqib et al., 2006).
- In alfalfa (lucerne), symptoms range from a yellow to red discoloration of the leaves, a yellowish-brown root discoloration under the periderm to plant death (Pilkington et al., 2003).

Note: While phytoplasma infections are usually detrimental to plant growth, some plants exhibit minor symptoms or are symptomless.

Pest Importance

Diseases caused by *Ca.* P. australiense impact economically important food and ornamental crops. Researchers have documented vineyard losses as high as 13%. Severely affected grape vines can produce up to 54% less fruit than healthy grape vines (CABI, 2004; NPAG, 2007). Papaya dieback is responsible for annual plant losses of 10% and up to 100% epiphytotics (epidemic among plants of a single kind over a wide area) in central and southern Queensland plantations (Glennie and Chapman, 1976; Guthrie et al., 1998). Australian lucerne yellows has caused a reduction in see yield, which has led to the cutting or plowing-under of seed crops, resulting in estimated losses of $7 million annually (Pilkington et al., 1999).

Establishment of *Ca.* P. australiense in the United States could impact trade because some countries, like Morocco, classify the pathogen as a dangerous quarantine pest (NPAG, 2007).

Known Hosts

Research implicates *Ca.* P. australiense as the cause of disease on the following hosts:

*Carica papaya (*papaya), *Catharanthus roseus* (periwinkle), *Cicer arietinum* (chickpea), *Coprosma robusta* (coprosoma), *Cordyline australis* (cabbage tree), *Cucumis myriocarpus* (paddy melon), *Curcurbita maxima* (pumpkin), *Cucurbita moschata* (pumpkin), *Exocarpus cupressiformis* (cherry ballart), *Fragaria ananassa* (strawberry), *Gomphocarpus fruticosus* (cottonbush), *Jacksonia scoparia* (winged broom pea), *Liquidamber styraciflua* (sweetgum), *Medicago sativa* (alfalfa), *Paulownia fortunei* (paulownia), *Phaseolus vulgaris* (bean), *Phorium tenax* (New Zealand flax), *Prunus persica* (peach), *Rubus ursinus* (boysenberry), *Trifolium pretense* (red clover), *Vigna radiata* (mung bean), and *Vitis vinifera* (grape) (EPPO, 1998; Liefting et al., 1998; White et al., 1998; Schneider et al., 1999; Wood et al., 1999; Padovan et al., 2000; Andersen et al., 2001; Davis et al., 2003; Beever et al., 2004; Bayliss et al., 2005; Gera et al., 2005; Jones et al., 2005; Lucas, 2005; Saqib et al., 2005; Steten and Gibb, 2005; Streten et al., 2005; Habili et al., 2006; Saqib et al., 2006; Getachew et al., 2007).

Known Vectors (or associated insects)

The only known vector of *Ca.* Phytoplasma australiense is *Oliarus alkinsoni* (not known in the United States). This vector is associated with phormium yellow leaf, also caused by *Ca.* P. australiense. Other vectors are suspected but not currently confirmed.

Known Distribution

Ca P. australiense is present in New Zealand and Australia. It is also present in Israel and Bolivia.

Potential Distribution within the United States

Ca. P. australiense is considered an imminent threat that could be introduced into the United States with imported leaves, stems, and roots of infected plants (NPAG, 2007).

Survey

Preferred Method: Visual inspection for symptoms associated with the phytoplasma. Several of the known symptoms should be found together before suspecting a phytoplasma infection on grape (CABI, 2004). Symptoms begin to appear in late spring and increase in incidence until January/February. Beyond this time, AGY symptoms begin to disappear as symptomatic leaves and shoots fall from the vine (CABI, 2004). Phytoplasma DNA was detected from most plants when samples were collected in January and February compared to those sampled in Oct-Dec. and March-May (Gibb et al., 1999). The disease appears most often in Chardonnay and Riesling grapes but has also been reported in other cultivars (Magarey and Watchel, 1986).

Key Diagnostics

A ‘universal’ polymerase chain reaction (PCR) assay has been developed that enables amplification of the 16S rRNA genes of phytoplasmas (Deng and Hiruki, 1991; Ahrens and Seemulller, 1992; Lee et al., 1993; Smart et al., 1996; Gibb et al., 1999). Following this amplification, the PCR product can be readily visualized after agarose gel electrophoresis by UV transillumination after staining with ethidium bromide. In addition, digestion of the PCR product with selected restriction endonuclease enzymes provides a DNA fingerprint in the form of 16S rDNA fragment patterns that can be used to determine phytoplasma identity (Gibb et al., 1999). Alternatively, PCR assays have been developed to detect only the AGY phytoplasma in grape (Davis et al., 1997; Gibb et al., 1999).

Easily Confused Pests

AGY resembles flavéscence dorée, bois noir, Goldgelbe Vergilbung, leaf curl, berry shrivel and other grapevine diseases (tomato big bud and an uncharacterized disease also believed to be caused by a phytoplasma) (Magarey and Wachtel, 1985; Davis et al., 1997; Lee et al., 1998; Constable et al., 1998; Gibb et al., 1999). Detection of flavéscence dorée by cloned DNA probes was accomplished by Daire et al. (1992).

Mechanical disruption to the phloem of grape shoots can cause symptoms similar to those associated with *Ca* P. australiense infection (CABI, 2004). It is important to inspect symptomatic shoots for damage to the vascular tissue due to breakage, restrictions of the vascular tissue due to tendrils or string wrapping tightly around shoots, and damage to the vascular tissue by boring insects.

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Phellinus noxius

Scientific Name
Phellinus noxius (Corner) G. Cunn.

Synonyms:
Corticum spp., *Fomes noxius*, *Hymenochaete noxia*, *Hymenochaete noxius*, *Phellinidium noxium*, *Poria setulalsocrocea*

Common Name
Brown root rot, brown cocoa root rot, brown root, brown tea root disease, collar rot, stem rot

Type of Pest
Fungus

Taxonomic Position
Class: Basidiomycetes, **Order:** Hymenochaetales, **Family:** Hymenochaetaceae

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

Pest Description
The basidiocarp, also referred to as a sporocarp or conk, is perennial, solitary or imbricate, sessile with a broad basal attachment, commonly resupinate. **Basidiocarps are not always produced in nature but can be induced in the laboratory** (Bolland et al., 1984). Pileus 5-13 x 6-25 x 2-4 cm, applanate, dimidiate or appressed-reflexed; upper surface deep reddish-brown to umbrinous, soon blackening, at first tomentose, glabrescent, sometimes with narrow concentric zonation, developing a thick crust; margin white then concolorous, obtuse. Context up to 1 cm thick, golden brown, blackening with KOH, silky-zonate fibrous, woody. Pore surface grayish-brown to umbrinous; pores irregular, polygonal, 6-8 mm, 75-175 µm diameter, dissepiments 25-100 µm thick, brittle and lacerate; tubes stratified, developing 2-5 layers, 1-4 mm to each layer, darker than context, carbonaceous.

Basidiospores approximately 4 x 3 µm, ovoid to broadly ellipsoid, hyaline, with a smooth, slightly thickened wall, and irregular guttulate contents. Basidia 12-16 x 4-5 µm, short clavate, 4-spored. Setae absent. Setal hyphae present both in the context and the dissepiment trama. Context setal hyphae radially arranged, up to 600 x 4-13 µm, unbranched or rarely branching, with a thick dark chestnut brown wall and capillary lumen; apex acute to obtuse, occasionally nodulose. Tramal setal hyphae diverging to project into the tube cavity, 55-100 x 9-18 µm, with a thick dark chestnut-brown wall (2.5-7.5 µm thick) and a broad obtuse apex. Hyphal system dimitic with generative and skeletal hyphae, non-agglutinated in the context, but strongly agglutinated in the dissepiments. Generative hyphae 1-6.5 µm diameter, hyaline or brownish, wall thin to somewhat thickening, freely branching, simple septate. Skeletal hyphae 5-9 µm diameter, unbranched, of unlimited growth, with a thick reddish-brown wall (up to 2.5 µm thick) and continuous lumen, non-septate (Pagler and Waterson, 1968).

Biology and Ecology

Phellinus noxius is a facultative parasite. It obtains nutrients from living or dead plants. As a tropical plant pathogen, its mycelium grows best at 25-30°C (77-86°F); it does not grow at temperatures below 4°C (39°F) or above 40°C (104°F). Colonies grown in the laboratory have distinctive raised brown and white plaques (patches) (Fig. 1) and occasionally produce arthrospores, which are asexual spores formed by the division of special hyphae into one-celled segments. Many fungi produce asexual spores, which allows dissemination of the fungus within the same plant or from plant to plant. Arthrospores of *P. noxius* have not been observed in nature, however, and their importance remains unknown (Brooks, 2002).

Figure 1. Mycelia (left) and arthrospores (right) of *P. noxius*. Photos courtesy of Pao-Jen Ann.

The name brown root rot refers to a brown to black mycelial crust formed by the fungus on the surface of infected roots and stem bases (Chang and Yang, 1998). However, *P. noxius* is generally considered a white rust fungus because of its ability to degrade lignin, a complex molecule that gives wood much of its strength and brown color (Fig. 2) (Chang and Yang, 1998). The mycelium of *P. noxius* also forms microhyphae and hyphae with extracellular sheaths. These structures are found where wood is being destroyed but their exact function is not yet known (Brooks, 2002).

Figure 2. Dry, honeycombed, white wood rot caused by *P. noxius*. Photo courtesy of Fred Brooks.

P. noxius is spread in two main ways. The first is by windborne spores (basidiospores), which can infect freshly cut tree stumps or fresh wounds. The second and most important is by root-to-root contact. The leading edge of the mycelial sleeve will infect healthy roots of other trees if they touch. Infected root pieces can remain viable for many years in the soil (Bolland, 1984; CABI, 2004). Basidiospores, the sexual spores, of *P. noxius* contain one nucleus (monokaryotic) and are haploid. When haploid hyphae (n) of different mating types touch each other, they can fuse to form a dikaryotic mycelium with cells containing two haploid nuclei (n + n). This dikaryotic mycelium is the most common condition in nature. The hyphae can be modified (swelling, thickening, or gluing together) to produce a mycelial crust or basidiocarps, which often start as small round patches on stems of dead trees. The patches may continue to grow flat against the wood, grow out into a shelf-like conk, or a combination of both. The upper surface of the basidiocarp is sterile, the lower fertile surface covered with small tubes or pores, These tubes are lined with basidia, whose two nuclei fuse and undergo meiosis, producing four haploid basidiospores. During wet weather, basidiospores are released and spread by the wind. Basidiospores may be responsible for some long distance dispersal of the fungus (Brooks, 2002).

P. noxius is present in native tropical forests and plantations on infected roots, stumps, and woody debris. It does not form survival structures such as sclerotia or resting spores but may persist in dead roots and colonized wood for many years (Chang, 1996). Centers of disease radiate outward as roots of healthy trees come in contact with roots of diseased trees. Trees of all ages are susceptible. A mycelial crust forms around infected roots, secreting wood-rotting enzymes as it moves up the roots to the stem. Crusts 0.5-1.0 cm thick usually extend 1-2 m (3-6 feet) up the stem, though crusts almost 5 m (15 ft) high have been measured. If the tree does not die from severe root rot, it is killed when the crust surrounds the stem and the underlying mycelium destroys the sapwood.

Figure 3. Mycelial crust of *P. noxius* on multi-trunked tree in the rainforest. Photo courtesy of Fred Brooks. www.bugwood.org

Symptoms/Signs

Symptoms of brown root rot are similar to those caused by other root rot pathogens: slow plant growth, yellowing and wilting of leaves, defoliation, branch dieback, and plant death (Brooks, 2002). Although dead wood is initially discolored reddish brown, it later becomes white, dry, and crumbly.

Figure 4. With age, reddish hyphae in wood agglutinate (stick together), turn black and brittle. Photo courtesy of Fred Brooks.

Signs of the pathogen, unlike the symptoms, are distinctive for this disease. *P. noxius* forms a thick, dark brown to black crust of mycelium around infected roots and lower stems (Fig. 3), which gives the disease its name. The leading edge of the crust is creamy white, glistens with drops of clear, brownish exudate, and is usually noticeable even in the dark understory of the rainforest. Patches of white mycelium are present between the bark and sapwood. As colonization progresses, white, soft, crumbly wood becomes laced with reddish strands of fungus hyphae that turn black with age (Fig. 4). Basidiocarps, or fruiting bodies, are purplish brown bracts (conks) with yellow-white growing margins and concentric blackish zones towards the edges (CABI, 2004). The basidocarps are gray to brown on the spore-forming surface (Brooks, 2002). Unlike other similar fungi, there are no rhizomorphs. Spread is by physical contact with the root encrustations.

Pest Importance

P. noxius is known to cause extensive damage in a wide range of species, including common species used in industrial forest plantations, commercial fruit orchards, commodity crops such as cacao and rubber, and non-commercial components of native forests (Hodges, 2005). The fungus would be expected to cause similar losses if introduced into the tropical and subtropical regions of North America.

Due to the extremely diverse host range and geographical distribution, the economic impact of *P. noxius* is highly variable (CABI, 2004). The impact can vary from insignificant to losses of 60% in rubber tree plantations after 21 years (Nandris et al., 1987). *P. noxius* has caused 10% mortality in plantations of *Swietenia macrophylla* in Fiji and 50% mortality in a three-year-old plantation of *Pinus merkusii* in Indonesia (Bolland, 1984).

Brown root rot has become one of the most serious problems of fruit and forest trees in central and southern Taiwan at elevations less than 800 m (Chang and Yang, 1998).

Known Hosts

Chang and Wang (1998) showed that *P. noxius* has a broad host range that includes woody hardwoods, conifers, and annual herbaceous plants. It seems, however, to be more saprotrophic than parasitic on annual herbaceous plants, because it does not cause severe wilting of host foliage. More than 200 woody plant species representing 59 families have so far been recorded as host plants for *P. noxius.* Hosts were not classified as major or minor hosts, because most hosts were reported simply as hosts with no attempt to classify economic loss or susceptibility to *P. noxius.*

Acacia spp. (wattles)*, Actinidaphne pedicellata (litsea), Adenanthera pavonina* (coralwood), *Agathis palmerstonii* (kauri gum), *Albizaia* spp. (albizia), *Aleurites* spp. (Indian walnut), *Alstonia scholaris* (blackboard tree), *Annona* spp. (sour sop, custard apple), *Aralia elata* (Japanese aralia), ***Araucaria cunninghamii*** (colonial, hoop pine), *Araucaria* spp., *Ardisia sieboldii, Areca triandra* (wild areca palm), *Artemisia capillaris* (wormwood), *Artocarpus* spp. (breadfruit), *Averrhoa carambola* (carambola), *Barringtonia* spp., *Bauhinia* spp. (bauhinia), *Bischofia javanica* (autumn maple tree), *Blepharocarya involucrigera* (bolly gum), *Bombax ceiba* (silk cotton), *Bougainvillea* spp. (bougainvillea), *Breynia nivosa* (snowflower), *Broussonetia kazinoki* (small paper mulberry), *Broussonetia papyrifera* (paper mulberry), *Calophyllum inophyllum* (Indian beauty leaf), *Cajanus cajan* (pigeon pea), *Camellia japonica* (camellia), ***Camellia sinensis*** (tea), *Canangia odorata* (ilang-ilan), *Cassia* spp., *Castanospora alphandii*, *Castenospermum australe*, *Casuarina* spp., *Cedrella mexicana, Ceiba pentandra, Celtis sinensis, Chorisia speciosa* (floss silk tree), *Chrysalidocarpus lutescens* (yellow areca palm), ***Cinnamomum camphora*** (camphor), *Cinnamomum* spp.*,* ***Cinnamomum zeylanicum*** (Ceylon cinnamon)*,* ***Citrus* spp**., *Cocos nucifera* (coconut), *Codiaeum variegatum* (croton), *Coffea* spp. (coffee), *Clutia* spp., *Cordia alliodora,* ***Cordia dichotoma*** (cordia)*, Crataegus* spp. (hawthorne)*, Crescentia cujete* (calabash)*, Crossostylis biflora, Crotalaria micans* (cascabelitos)*, Cryptocarya* spp. (crytocarya), *Cupressus lusitanica*, *Cycas taiwaniana* (Taiwan cycas), *Dalbergia sissoo* (sissoo tree), *Delonix regia* (flame tree), *Dimocarpus longan* (longan), *Diospyros* spp.*, Duranta repens* (creeping sky flower), *Dysoxylum samoense* (maota), *Elaeis guinennsis* (African oil palm), *Elaeocarpus serratus* (Ceylon olive), *Elaecarpus sylvestris* var. *ellipticus*, *Eribotrya japonica* (loquat), *Erythrina variegata* (Indian coral tree), *Eucalyptus* spp., *Ficus* spp., *Fraxinus formosana* (island ash), *Garcinia mangostana* (mangosteen), *Gardenia jasminoides* (cape jasmine), *Grevillea robusta* (silver oak)*, Hedera australiana, Heritiera* spp., ***Hevea brasilensis*** (rubber tree)*, Hibiscus* spp. (hibiscus), *Hydrangea chinensis* (Chinese hydrangea), *Ilex rotunda* (kurogane-mochi)*, Ipomoea pescaprae*, *Kigelia pinnata* (sausage tree), *Koelreuteria henryi* (flame gold rain tree), *Lactuca indica* (wild lettuce), *Lagerstroemia* spp. (crape myrtle), *Lantana camara* (lantana), *Leucaena leudocephala* (white popinac), *Ligustrum japonicum*, *Liquidambar formosana* (sweetgum)*, Litchi chinensis* (litchi)*, Mangifera indica* (mango), *Melaleuca leucadendron* (cajuput tree), *Melia azedarach* (China berry), *Melicope merrilli* (melicope), *Melodinus augustifolius* (narrow leafed melodinus), *Michelia* spp. (michelia), *Morus australis*, *Muntingia calabura* (Indian cherry), ***Murraya paniculata*** (orange jasmine), *Nandina domestica* (sacred bamboo), ***Nerium oleander*** (oleander), *Ochroma* spp. (balsa), *Pachira macrocarpa* (Malabar chestnut), ***Persea americana*** (avocado),

Pinus* spp.** (pine), *Piper nigrum* (black pepper), *Pistacia chinesensis* (Chinese pistachio), *Podocarpus macrophyllus* (yew), *Pongamia pinnata* (pongamia), *Prunus persica* (peach), *Pterocarpus indicus* (rose wood), *Pygeum tuberanum*, *Pyrus pyrifolia* (pear), *Osmanthus fra*grans (sweet osmanthus), *Rhaphiolepis indica* var. *umbellata*, *Rhodendron obtusum* (rhododendron), *Rhus succedanea* (Japanese waxtree), ***Rosa* spp.** (rose), *Roystonea regia* (royal palm), *Salix babylonica* (willow), *Sauramja oldhami*, *Schefflera* spp. (scheffera), *Stenocarpus sinuatus*, *Sterculia* spp.(sterculia), *Swietenia* spp. (mahogany), *Syzygium samarangense* (wax apple), *Tabebuia chrysantha* (yellow golden bell tree), ***Tectonia grandis (teak), *Terminalia* spp. ***Theobroma cacao*** (cocoa), *Trema orientalis*, *Ulmus parviolia* (Chinese elm), ***Vitis vinifera*** (grape). For a complete host listing see: Bolland, 1984; Neil, 1986; Almonicar, 1992; Ann et al., 2002; CABI, 2004; Sahashi et al., 2007.

Known Vectors (or associated insects)

P. noxius does not have a known vector or associated organisms.

Known Distribution

The fungus is confined mainly to tropical areas of the world.
Asia: India, Indonesia, Japan, Malaysia, Myanmar, Pakistan, Philippines, Singapore, Sri Lanka, Taiwan, and Vietnam. **Africa:** Angola, Benin, Burkina Faso, Cameroon, Central African Republic, Congo, Cote d`Ivoire (Ivory Coast), Gabon, Ghana, Kenya, Nigeria, Sierra Leone, Tanzania, Togo, and Uganda. **Central America:** Costa Rica, Cuba, and Puerto Rico. **South America:** Brazil, French Guiana. **Oceania:** American Samoa, Australia, Federated states of Micronesia, Fiji, Guam, New Caledonia, Niue, Northern Mariana Islands, Papua New Guinea, New Zealand, Samoa, Solomon Islands, and Vanuatu.

Potential Distribution within the United States

P. noxius has a broad host range and would undoubtedly find numerous suitable hosts in North America, but would most likely be restricted to tropical or near tropical regions (Hodges, 2005).

Survey

<u>Preferred Method:</u> Visual survey is the most common method used to survey for *P. noxius.* A dark brown mycelial mat or sleeve on the surface of the roots and up to the base of the stem is used reliably for field identification of *P. noxius*. Soil is scraped away around the collar and the main roots and the distinctive mycelial sleeve is often present (Nandris et al., 1987). Particular attention should be paid to trees that appear wilted or dead. *P. noxius* tends to be a problem in cleared forests converted to agricultural land (tree farms) or in disturbed areas and surveys should be conducted in these areas.

<u>Alternative Method:</u> Early detection of the pathogen before typical wilt symptoms are visible is very difficult and time consuming. Baiting out the pathogen by placing sticks of a susceptible host in the soil and retrieving for laboratory examination after 3 weeks is also conducted, particularly in virgin forests to detect parasites on the root system of wild trees (Nandris et al., 1987; CABI, 2004). According to Nandris et al. (1987), the

area around the root collar can be mulched for 3 weeks to provide a damp zone that allows the superficial mycelium to progress from the roots onto the trunk of rubber trees. When the mulch is removed, the mycelial filaments of the pathogen can be observed.

Key Diagnostics

According to Ann et al (1999), after plating surface sterilized diseased root tissue on potato dextrose agar amended with ampicillin and benomyl, the cultural and morphological characteristics are the fungus are examined and compared. The Key of the Polyporaceae described by Cunningham (1965) is then used for identification of the fungus. In culture, mycelia are initially white and then brown with irregular dark brown lines or patches. In addition, staghorn-like hyphae and arthrospores, but no clamp connections are commonly observed (Sahasi et al., 2007).

Chang (1995) developed a selective medium for *P. noxius* using malt extract agar as a basal medium amended with benomyl, dicloran, ampicillin, and gallic acid. Tergitol NP-7 was added for isolation from soil.

Bolland et al. (1984) developed a method to induce sporulation in basidiocarps of *P. noxius* to obtain single spore isolates.

Easily Confused Pests

P. noxius basidiocarps are sometimes confused with *P. lamaensis*, another tropical *Phellinus* species. *P. lamaensis* sporocarps have short, reddish-brown, cone-shaped cells called hymenial setae growing into their pores, however, *P. noxius* does not.

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Xylella fastidiosa (citrus variegated chlorosis strain)

Scientific Name
Xylella fastidiosa Wells

Common Name(s)
Amarelinho (Brazil), pecosita (Argentina)

Type of Pest
Plant pathogenic bacterium

Taxonomic Position
Class: Proteobacteria, **Order:** Xanthamonadales, **Family:** Xanthamonadaceae

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

Pest Description
*Xylella fastidios*a is a fastidious, gram-negative, rod-shaped, xylem-limited bacterium with rippled cell walls. It is strictly aerobic and non-flagellate, does not form spores, and measures 0.4 x 4 μm (Hartung et al., 1994). The bacteria are tightly packed in the lumen of xylem vessels when viewed by electron microscopy (Fig. 1) and are transmitted in a persistent manner by various leafhopper species. The vessels are ultimately blocked by bacterial aggregates and by tyloses and gums formed by the plant.

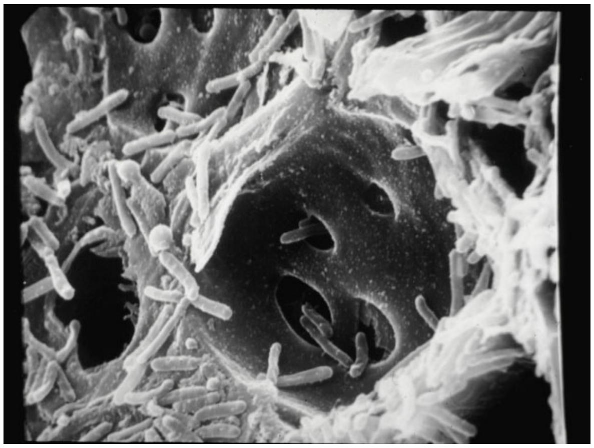

Figure 1. *X. fastidiosa* in xylem vessels. Photo courtesy of D.R. Cook

This taxon currently includes at least three different pathogen groups/strains. Bacteria in one group, the group of greatest concern, are known to infect citrus (citrus variegated chlorosis) and coffee (coffee leaf scorch) in South America and were recently found in Costa Rica. Another strain infects plum in South America (plum leaf scald). The other groups occur in North America, where the diseases they cause have been known for many years. The bacteria in the North American groups infect many plants, including grapes (Pierce's disease), alfalfa (alfalfa dwarf), peach (phony disease), almonds (almond leaf scorch), and cause scorch diseases of a number of different shade trees (sycamore, oak, and maple). One distantly related strain causes pear leaf scorch in Taiwan (Horvath, 2005).

Biology and Ecology
Epidemiological studies suggest that most spread of citrus variegated chlorosis (CVC) (Fig. 2) is from tree to tree within citrus groves. The bacterium is spread from plant to plant by grafting with infected bud material, by natural root grafts, and by sharpshooter leafhoppers (Hemiptera Cicadellidae). Because the bacterium is restricted to the xylem, transmission through budwood is low but nevertheless sufficient for the widespread distribution of the disease. In Brazil, 12 of 16 sharpshooter species tested were able to transmit the bacterium (Lopez, 2000). In the United States, the blue-winged sharpshooter (*Oncometopia nigricans*) has been shown experimentally to be a vector of the CVC strain of *X. fastidiosa* (Brlansky et al., 2002). The glassy winged sharpshooter (GWSS) (*Homalodisca coagulata*) has been observed transmitting oleander leaf scorch (another *X. fastidiosa* strain) with more than 80% efficacy. Although at a low level, Damsteegt et al. (2006) showed transmission of the CVC strain of *X. fastidiosa* by the GWSS. The bacterium has recently been shown to be seedborne, which may require the monitoring and testing of seed and budwood sources and limiting the movement of fruit from infected locales (Li et al., 2003).

Pierce's disease, caused by another strain of *X. fastidiosa*, already limits grape production in the southeastern United States and Central America. **Citrus and coffee strains of *X. fastidiosa* from Brazil have been shown experimentally to induce Pierce's disease of grapevine (Li et al, 2002). Therefore, any future introduction of the CVC strains of *X. fastidiosa* into the United States would pose a threat to both the sweet orange and the grapevine industries, given the high populations of sharpshooters found in many areas of Florida and California.** The Pierce's disease strains of *X. fastidiosa* apparently are not capable of inciting CVC. Citrus has been grown commercially in California and Florida for more than a century in the presence of Pierce's disease-infected grapes, and CVC has never been reported (Li et al., 2002).

Figure 2. Citrus variegated chlorosis . Photo courtesy of M.J.G. Beretta

Symptoms/Signs
Early symptoms of CVC (Fig. 3) include a foliar interveinal chlorosis (yellowing) resembling zinc deficiency on the upper surface of young leaves as they mature. As the leaves mature, small light-brown gummy lesions develop on the lower surface,

corresponding with the chlorotic areas on the upper surface. The lesions become dark-brown or even necrotic, enlarging with time. The leaves are often smaller than normal. Very young leaves do not show symptoms. The English name for the citrus disease, CVC, comes from the striking chlorotic variegation induced on sweet orange leaves by the pathogen.

Figure 3. Leaf interveinal chlorosis. Photos courtesy of A. Purcell and R.F. Lee.

Initially, symptoms are present in some branches but the entire canopy displays the disease over time. Young, non-fruit bearing trees become systemically infected more rapidly and generally display more severe symptoms than older trees. Trees more than 8 to 10 years old are not usually totally affected, but rather have symptoms on the extremities of branches. Infected trees show stunting and reduced growth rates, with twig and branch dieback and canopy thinning.

Symptoms of CVC on grape would most likely resemble Pierce’s disease (Fig 4). Symptoms of Pierce's disease first appear as water stress in midsummer, caused by blockage of the water-conducting system by the bacteria. The occurrence of the following four symptoms in mid- to late summer indicates the presence of Pierce's disease: (1) leaves become slightly yellow or red along margins in white and red varieties, respectively, and eventually leaf margins dry or die in concentric zones; (2) fruit clusters shrivel or raisin; (3) dried leaves fall leaving the petiole (leaf stem) attached to the cane; and (4) wood on new canes matures irregularly, producing patches of green, surrounded by mature brown bark. Delayed and stunted shoot growth occurs in spring following infection even in vines that did not have obvious symptoms the preceding year.

Usually only one or two canes will show Pierce's disease symptoms late in the first season of infection, and these may be difficult to notice. Symptoms gradually spread along the cane from the point of infection out towards the end and more slowly towards the base. By mid-season some or all fruit clusters on the infected cane of susceptible varieties may wilt and dry. Tips of canes may die back, roots may also die back. Vines of susceptible varieties deteriorate rapidly after appearance of symptoms. Shoot growth of infected plants becomes progressively weaker as symptoms become more pronounced.

One of the challenges in identifying this pathogen results from the relatively long incubation period between infection and resulting symptoms, which results in latent infections. In citrus, typical symptoms do not develop until 9 to 12 months after infection in the field and symptoms can easily be mistaken for a zinc deficiency problem. Symptom expression and incidence of CVC appear to be greater in warmer climates.

Figure 4. Symptoms of Pierce's disease on grape. Marginal scorching is preceded by concentric reddening and chlorosis. Note: bare leaf petioles. Photo courtesy of A. Purcell.

Pest Importance

Citrus variegated chlorosis, a new disease affecting primarily sweet orange (*Citrus sinensis*), has been associated with strains of *X. fastidiosa* in Brazil (Chang et al., 1993; Hartung et al., 1994). The disease was first observed in Argentina during the early 1980s and soon thereafter in Brazil (Roberto et al., 2002). The author's initial reports suggested that the disease posed an immediate threat to the Brazilian and world citrus industry. In 1996, the Brazilian state of Sao Paulo was responsible for 83% of the national citrus production and CVC was present in all citrus growing areas of Sao Paulo (Lopes et al., 2000). In the year 2000, 35% of the 200 million sweet orange trees in Sao Paulo showed CVC symptoms, representing a direct loss of more than US $100 million (Li et al., 2002). It has been estimated that in the absence of remedial measures a CVC incidence of 90% in a grove could occur 12 years after introduction of a single infected tree and that an individual tree may become unproductive within three years (Gottwald et al., 1993). The severity of the CVC disease problem contributed to the selection of a citrus strain of *X. fastidiosa* as the first plant pathogenic bacterium to have its entire genome sequenced (Simpson et al., 2000).

Known Hosts

The primary host of the CVC strain of *X. fastidiosa* is citrus. All sweet orange varieties are highly susceptible, with limes and grapefruit being less susceptible. The sweet orange cultivars Pera, Hamlin, Natal, and Valencia appear to be extremely susceptible to CVC. Lemons, mandarins, and some mandarin hybrids range from susceptible (show leaf symptoms) to tolerant (very mild or no leaf symptoms) to resistant (no leaf symptoms and no detectable bacteria). Coffee (*Coffea arabica* and *C. canephora* var. *robusta*) plants are susceptible, and CVC bacteria cause the disease known as coffee leaf scorch. Several scientists have suggested that coffee may be the original host of this strain, with the bacteria later being moved by vectors and adapting to citrus in nearby plantings.

Many strains of *X. fastidiosa* have wide natural host ranges, and alternate hosts may serve as a major source of inoculum. The following (symptomless) weeds were collected in citrus groves in Brazil and found to be naturally-infected with the CVC strain of *X. fastidiosa* by polymerase chain reaction (PCR) testing: *Alternanthera tenella, Commelina benghalensis, Bidens pilosa, Euphorbia hirta, Brachiaria decumbens*., *Cenchrus echinatus, Digitaria horizontalis, Digitaria insularis, Spermacoce latifolia,* and *Solanum americanum* (Horvath, 2005). The following plants were shown to be hosts when mechanically inoculated: grape (*Vitis vinifera*), Madagascar periwinkle (*Cantharanthus roseus*), tobacco (*Nicotiana tabacum*), alfalfa (*Medicago sativa)*, and the weeds *Brachiara plantaginea* and *Echinochloa crus-galli* (Li et al, 2002, Lopes et al., 2000, Lopes et al., 2003, Monteiro et al., 2001).

Known Vectors (or associated insects)

In Brazil, 12 of 16 sharpshooter species tested were able to transmit the bacterium. In the United States, the blue-winged sharpshooter (*Oncometopia nigricans*) has been shown experimentally to be a vector of the CVC strain of *X. fastidiosa*. The glassy winged sharpshooter (GWSS) (*Homalodisca coagulata*) has been shown to transmit the CVC strain of *X. fastidiosa.*

Known Distribution

South America: Argentina, Brazil, Costa Rica, Paraguay

Potential Distribution within the United States

Citrus production areas with large populations of the sharpshooter vectors would be at considerable risk, particularly Florida and California. In Florida, the sharpshooter *Oncometopia nigricans* is native, feeds on citrus, and is capable of transmitting *X. fastidiosa* (Brlansky et al., 2002). Studies have also shown that the GWSS, which is present in the southeastern United States and California, can transmit the CVC bacterium (Damsteegt et al., 2002). Recent epidemiological data showed that proximity to citrus increased the incidence and severity of Pierce’s disease of grapevines in the

Temecula Valley of California (Perring et al., 2001). This relationship occurs because in California the GWSS preferentially feeds and reproduces to high levels on citrus.

Survey

Preferred Method: Survey for CVC will involve a visual inspection of grape. Surveys for visual symptoms will need to be made in vineyards. Methods are currently described for surveying citrus in the dooryard, commercial grove, and nursery plantings (Horvath, 2005), however, little information exists for grape. Vineyards within 1/2 to 1 mile of citrus or avocado groves appear to be at greatest risk. Infected vines may occur randomly throughout the vineyard making it necessary to do a walkthrough and visually inspect all vines for the presence of symptoms. Regardless of the inspection method, diseased tissue needs to be collected and an ELISA test be conducted that will assess if *X. fastidiosa* is present. Additional confirmation will need to occur via PCR to determine if the CVC strain is present.

Key Diagnostics

Classical procedures for diagnosis of *X. fastidiosa* have been based on cultural characteristics, serology, electron microscopy and molecular techniques (Hartung et al., 1994). Recently a number of polymerase chain reaction (PCR) procedures have been developed for detection of *X. fastidiosa* from plant tissues and in the vector, including real-time PCR (Bextine et al., 2005; Bextine and Child, 2007). Primers have been developed that detect most, if not all, strains of *X. fastidiosa* (Firrao and Bazzi, 1994, Minsavage et al., 1994) and that are specific to the strain that causes CVC (Pooler and Hartung, 1995, Beretta et al, 1997).

Easily Confused Pests

Zinc deficiency may cause similar patterns of chlorosis in grape and citrus but are distinctively different in pattern to an experienced observer. Diseased plants do not respond to zinc fertilization. Water stress can also cause similar symptoms in grape. In grape, esca (France, Italy, Spain) or measles (United States), caused by three species of *Togninia,* has leaf scorching symptoms similar to Pierce's disease. Measles normally occurs early in the summer (June, July in Europe) following at least several days of hot weather, whereas Pierce's disease symptoms begin to appear after fruits begin to color (CABI, 2004).

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Secondary Pests of Grape (Truncated Pest Datasheet)

None at this time

Tertiary Pests of Grape (Name and Photo only) – on other lists; not a CAPS priority, but a potential threat to grape and exotic to the United States

Monilinia fructigena

Common name: brown rot

Reason for Inclusion in Manual

National Threat

Figure 1. Left: Signs and symptoms associated with *Monilinia fructigena* on peach. Right: Morphologies of cultures of *Monilinia laxa* (left), *M. fructicola* (right), and *M. fructigena* (bottom). Photos courtesy of http://www.biolib.cz/cz/image/id5445/?orderby=1&imgauthID=14 and APS Press, Compendium of Stone Fruit Diseases, respectively.

Pseudopezicula tracheiphila

Common name: red fire disease

Reason for Inclusion in Manual

National Threat

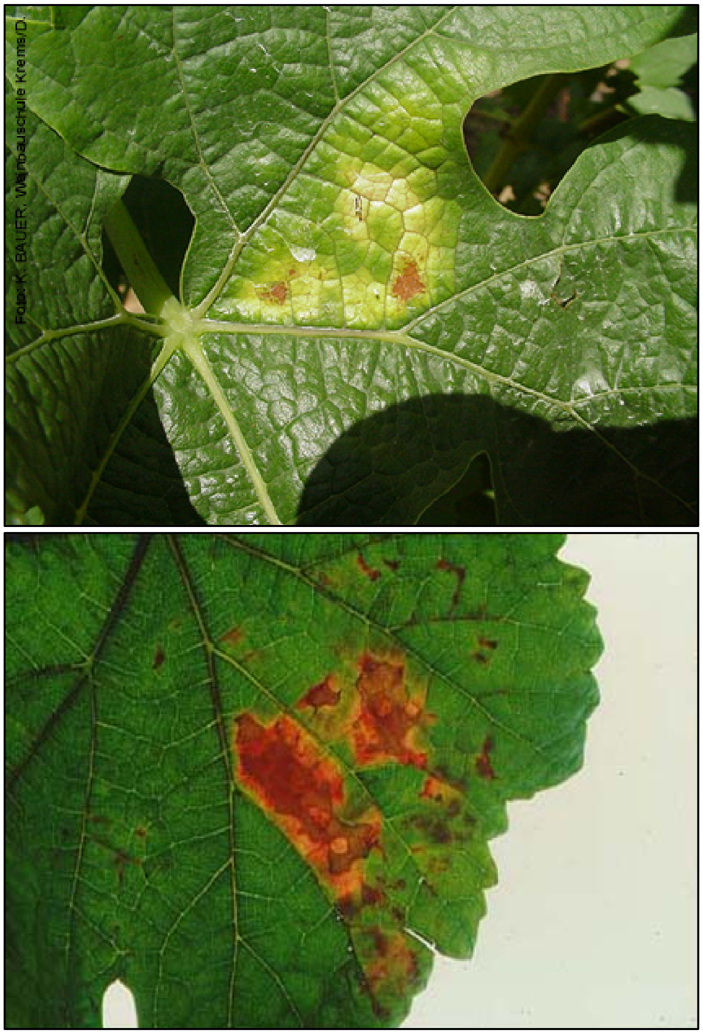

Figure 1. Symptoms are lesions on leaves, which are initially yellow on white-fruited cultivars (top) or bright red to reddish brown on red- and black-fruited cultivars (bottom). Later, a reddish brown necrosis develops. Photos courtesy of Karl Bauer and N. Berger, respectively.

Xylophilus ampelinus

Common name: bacterial blight of grapevine

Reason for Inclusion in Manual

National Threat

Figure 1. Symptoms on grapevine shoots and leaf. Photos courtesy of EPPO.

Abbreviated Table of Survey Techniques for Grape Pests

Arachnids, Insects, Mollusks

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Achatina fulica – giant African snail	Mollusk	*A. fulica* is easily seen due to its large size, and attacked plants exhibit extensive rasping and defoliation. The weight of the number of snails on a plant can break the stems of some host species. *A. fulica* can also be detected by signs of ribbon-like excrement, and slime trails on plants and buildings.	Preferred Method. Surveys are best carried out at night using a flashlight, or during overcast morning or evenings following a rain event. Look for the following clues that suggest the presence of snails: chewing damage to plants, eggs, juveniles and adults, empty snail shells, mucus and slime trails, large, ribbon-like feces, and an increase in rat population densities in an area.	Alternative method. Use traps to supplement a visual inspection, if time and resources allow. Use commercial brands of slug bait to attract snails; however, due to the slowacting effects of the molluscicide, these baits alone are not effective in trapping snails.	*Achatina fulica* remains active at a range of 9-29°C (48-84 °F). *Achatina fulica* begins hibernating at 2°C (35°F), and begins aestivation at 30°C (86 °F).

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Adoxophyes orana – summer fruit tortrix moth	Moth	Leaves may appear wilted, yellow, shredded, or dead. Leaves likely to be rolled or folded and held together with silk webbing. Lesions on new growth. External feeding damage and silk webbing in flowers. Fruit deformation (skin or general shape), scabs or pitting on fruit. Damage to stems, flowers, leaves, and fruit. Younger growth preferred, but damages all age plants. Fruit trees (Rosaceae) preferred host.	Alternative Method. Look on leaves, stems, and pods for: eggs on stems and leaves, external feeding on leaves and fresh growth. Feeding will deform leaves and create areas of dead tissue. Protected feeding sites (larvae bind together leaves, flower buds, or other plant parts with silk. Late instars may be found in the crown on new shoot growth. Silken pupal cocoons in leaves, on stems, or in old mummified fruit. Frass (excrement)	Preferred Method. Pheromone trap: Adoxomone, a 9:1 blend of (Z)-9-tetradecenyl acetate and (Z) – 11-tetradecenyl acetate, is attractive to this moth. For general monitoring and surveys, traps should be placed 45 m apart, approximately 1.5 m above the ground. This lure (9:1 blend) is available from the CPHST-Otis lab.	Summer: Adults present late May - late June (1st gen.), late July – early Sept (2nd gen.), October (3rd gen.)

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Aleurocanthus spiniferus – spiny black fly	Whitefly	Sticky honeydew deposits accumulate on leaves and stems and usually develop black sooty mold fungus, giving the foliage (even the whole plant) a sooty appearance. Infested leaves may be distorted. The insects are most noticeable as groups of very small, black spiny lumps on leaf undersides. Associated organisms: Ants may be attracted by the honeydew.	Preferred Method. Inspect for spiral egg masses and larvae on the underside of leaves. The adults may be found on tender terminal growth. Look for distorted leaves with immature stages of *A. spiniferus* on the undersides. The adults fly actively when disturbed. Good light conditions are essential for detection; in poor light, a powerful flashlight is helpful. Examine plants, especially shrubs or trees, closely for signs of sooty mold or sticky honeydew on leaves and stems, or ants running about. A heavy infestation gives trees an almost completely black appearance. A large hand lens may be necessary to help recognition of the dorsal spines on immature stages.	No trap available.	May be found throughout the year in tropical regions, but little breeding occurs during cold periods.

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. On grape, larvae scrape the skin and feed on the fruit contents. 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. 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 CPHST- Otis lab. 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
Brevipalpus chilensis – grape flat mite	Mite	Infested plant tissues become yellow, discolored and abnormally formed. New foliage & growing points are reduced in size, discolored, and formed abnormally.	Preferred Method. 15x Hand Lens All stages develop on underside of leaves, especially along the midrib vein of mature leaves. Look for adult females on grape bunches near pedicel and under bark or petiole cavity. External feeding on fruits & pods, growing points, leaves, and whole plant.	No trap available	Winter: Inactive on grapes. Spring: Loss of/ reduced size of new growth. Summer: Leaf rolling, leaf discoloration, fruit damage.
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
Conogethes punctiferalis – yellow peach moth	Moth	Small, irregularly spaced holes on seedlings. Larvae may bore into and feed on fruit/seeds. Characteristic dead hearts, reduced flowering. All plant parts are fed on internally. Fruits and pods may be discolored, with lesions and drop prematurely. Growing points may have evidence of boring, external feeding and have dead heart and mycelium present. Leaves may have webbing.	Alternative method. Scrape off bark in winter to look for overwintering cocoons. Look for dead hearts, irregularly spaced feeding holes, feces on outside of fruits, boring, and webbing on leaves. Adults nocturnal. 5 gen./year (China), 2-3 gen./yr (Japan).	Preferred method. Combination of synthetic female sex pheromones, (E)-10-hexadecenal and (Z)-10-hexadecenal. Males also emit sex pheromones. Ratios used vary widely. Light trap and sugar-vinegar traps attract moths but is not recommended.	Winter: Overwinter as 5th instar larvae in cocoons under bark. Spring: Larvae pupate within cocoons, moths emerge in mid-May (Japan) Summer: 1st gen. larvae attack peaches, later generations attack chestnut (Japan). Reported as late season pest of cotton (Australia).

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
Cryptoblabes gnidiella – citrus pyralid	Moth	Symptoms vary according to the feeding site, but the presence of silk, which indicates larval activity, is normally the obvious symptom of damage. It is often accompanied by frass produced by the larva. Associated with internal feeding on fruits and stems and external feeding on leaves. Associated organisms: Often associated with the honeydew produced by aphids and mealybugs.	Alternative method. Visual survey of larvae on leaves, in stems, or in fruit. Larvae are often found associated with infestations of other pests. On grape, for example, larvae are found following attack by *Lobesia botrana* (European vine moth).	Preferred method. A mixture of (Z)-11-hexadecenal, (E)-11-hexadecanal, (Z)-13-octadecanal, and (E)-13-octadenal on rubber septa has been found to attract *C. gnidiella* to traps. The commercial availability of this pheromone, however, is unknown at this time.	Adult moths were caught in pheromone traps in March-April (5%), June-Sept. (75%), and Oct.-Dec. (20%) in avocado orchards in Israel. More moths were trapped in young orchards than in mature orchards. Winter: Overwinter on fresh or dry fruits remaining on trees or vines.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Epiphyas postvittana – light brown apple moth	Moth	The insect will feed on foliage, flowers, and fruit. In spring, the pest feeds on new buds while later generations feed on ripened fruits. After the first molt they construct typical leaf rolls (nests) by webbing together leaves, a bud and one or more leaves, leaves to a fruit, or by folding and webbing individual mature leaves. During the fruiting season, they also make nests among clusters of fruits, damaging the surface and sometimes tunneling into the fruits. Fruit surface feeding is common within larval nest sites and is typically caused by later instars. Clusters of fruit are particularly susceptible. On a fruit, the calyx offers protection from parasitoids and is the best feeding location for young larvae. Larvae entering the fruit through the calyx may cause internal damage. Wet conditions may allow the entry of rot organisms. Feeding on the foliage by larvae causes ragging and curling of the foliage. Associated organisms: Larvae have been shown to introduce *Botrytis cinerea* in wounds when contaminated with spores.	Alternative Method. Used to monitor population dynamics of eggs and larvae. In grape, 40 vines were inspected per sampling date. Egg masses most likely to be found on leaves. Larvae most likely to be found near the calyx or in the endocarp; larvae may also create "irregular brown areas, rounds pits, or scars" on the surface of a fruit. Larvae may also be found inside furled leaves, and adults may occasionally be found on the lower leaf surface	Preferred Method. Pheromone traps widely used. (*E*)-11-tetradecenyl acetate and (*E*,*E*)-(9,11) tetradecadienyl acetate key components in ratio of 20:1 are highly attractive to males. This lure is available from the CPHST-Otis lab in a 20:1 ratio.	Spring: 1st gen. larvae Summer: 2nd gen. larvae

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Eudocima fullonia – fruit piercing moth	Moth	For most moth pests, the larvae are the damaging stage. The fruit-piercing moth differs in this aspect, because it is the adult moth that is the damaging stage, and the larvae are essentially not harmful. The mouth parts of the moth are about an inch (2.5 cm) long and strong enough to penetrate through tough-skinned fruit. Once the moth has punctured the skin of the fruit, a process that usually takes a few seconds, it feeds upon the juices of the fruit. Feeding occurs at night and the fruit does not have to be ripe to be fed upon by this moth. Fruit flesh damaged by this moth becomes soft and mushy differing from fruit damaged by fruit flies, which is more liquid. A round, pinhole-sized puncture is made in fruits. The hole serves as an entry point for disease organisms and can result in early fruit drop. A small cavity is left in the fruit in the feeding site. The area of the fruit around the cavity will be dry and spongy.	Preferred Method. Moths are most active in the first few hours of the night. The large, red-glowing eyes of the moths are easily seen. The most effective way to monitor for fruit-piercing moths is to inspect the crop by torchlight after sundown beginning a few weeks before harvest. Surveys are typically begun 30 minutes after sundown and last one hour. Check trees/vines in the two outer rows of an orchard, particularly on the leeward side. Most damage occurs in the peripheral rows. Foliage of host plants may be inspected for larvae and other life stages.	No pheromones or semiochemicals have been identified for *E. fullonia*.	Summer: Adult moths likely to be found on mature fruit several weeks before harvest.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Eutetranychus orientalis – citrus brown mite	Mite	Leaves chlorotic (yellowed); younger, tender leaves when damaged show margins that are twisted upwards. Pale yellow streaks (stippling) along the midrib and veins initially, progressing to a grayish or silvery appearance of the leaves is present. Heavy infestations can cause leaf necrosis, leaf fall and plant dieback Damage to leaves. Attacks young and old plants.	Preferred method. Hardly visible to the naked eye appearing as small reddish or brown dots on the leaf surface. Shake an infested leaf above a sheet of white paper and use a small hand lens to observe behavior on the leaves. Check leaves that have the stippling effect of mite feeding. Look for: Eggs on upper leaf surface; Immature stages and adults on upper leaf surface; Webbing possible (often dust covered), providing protection for the eggs; Spread of the mite is windborne and new infestations commonly occur at the field perimeters; Field perimeters should be scouted especially field perimeters facing prevailing winds. Alfalfa may play role in dispersing tetranychid mites to other crops. Fields near alfalfa should be targeted.	No trap available.	Infestations can be observed any time during the growing season. Summer: The pest multiplies rapidly during hot and dry weather conditions.

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. African black beetles eat the cutting and rootlings at or just below ground level, ring barking the vine, and cause wilting and collapse. Damage is primarily in first two years after planting, as vines become too woody to be damaged by the beetle. 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
Homalodisca coagulate – glassy-winged sharpshooter (GWSS)	Moth	GWSS egg masses appear as small, greenish blisters. Eggs are covered with a white material scraped from deposits on the female forewings. These blisters are easier to observe after the eggs hatch, when they appear as tan to brown scars on the leaves. The excreta of the GWSS also can cause a whitewashed appearance on leaves, fruit and even on the sidewalk under it. The GWSS is large enough to be seen with the naked eye, it is very inconspicuous in nature. The brown coloration of the insect blends very well with the color of the twigs where it is usually found.	Preferred Method. Examine foliage for egg masses, juveniles, and or adults.	Preferred Method. Surveys in California are currently being conducted using a combination of sticky traps and visual surveys. Yellow sticky traps are commonly used for surveillance and detection of adults in orchards. Panels measuring a minimum of 5” x 9” are the trap of choice for GWSS.	The flight temperature threshold for GWSS is approximately 18 °C (65 °F). Trapping will not be effective during periods when temperatures are lower than this threshold. Trapping season begins no earlier than March 1 and ends October 31, depending on local conditions.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Lobesia botrana – European grape vine moth	Moth	On grape inflorescences (first generation), neonate larvae penetrate single flower buds. Symptoms are not evident initially, because larvae remain protected by the top bud. 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. Frass may remain adhering to the glomerules. On grapes, larvae feed externally and when berries are a little desiccated, they penetrate them, bore into the pulp and remain protected by the berry peel. Larvae secure the pierced berries to surrounding ones by silk threads in order to avoid falling. 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. May increase susceptibility to fungal or acid rot development.	Alternative method. 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. Adults fly mainly between the first rows of grapevines close to wind breaks and slopes facing the sun.	Preferred method. Pheromone-baited traps (*e.g.*, Pherocon 1C, Zoecon) have been used to monitor male flight activity and to make informed treatment decisions in grape production areas. A pheromone lure is available from the CPHST Lab, Otis. The lure 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. high. 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 egg laying in early spring. Adults tend to appear in vineyards when vines begin to flower,

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Oxycarenus hyalinipennis – cotton seed bug	True bug	*O. hyalinipennis* has been observed sucking the fruits of grapes. The feeding damage appeared as greasy spots that exuded light colored gum. Black feces were also present on the fruit.	Preferred Method. Visual inspection is the only survey method available at this time. Adult clusters have been observed on leaves of mango, guava, and citrus. Additionally, sweep netting of weeds between cotton rows or at field edges is recommended. Adults prefer crevices in such resting sites as tree trunks, undersides of leaves on trees, pods of legumes, dried flower heads, roots of grasses, under sheath leaves of corn and sugarcane, telephone poles or wooden posts, old nests of *Polistes* spp. (paper wasps), and crevices between strands of barbed wire.	Pheromones are not currently available for *O. hyalinipennis.*	*Oxycarenus hyalinipennis* begins feeding, mating, and egg laying when the seeds of their host become available.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Planococcus lilacinus – coffee mealybug	Mealybug	Symptoms on coconuts and cocoa are described as button nut shedding, drying of the inflorescence, and death of tips of branches. Dense colonies form conspicuous patches on fruits. Copious honeydew excretion may result in sooty mold development near colonies and the attraction of attendant ants. Fruits have been reported to have an abnormal shape and drop prematurely. Associated organisms: Ants may be attracted by the honeydew.	Preferred Method. *P. lilacinus* may be detected by thoroughly inspecting its normal habitats such as fruits, growing plant tips, shoots and roots. Fruits, plant parts and seedlings of suspect host plants should be thoroughly inspected, if necessary, under a hand lens. Males were said to pupate on the underside of leaves and to be scarce.	No trap available. No pheromones have yet been identified for *P. lilacinus*.	Infestations can be observed any time during the growing season.
Planococcus minor – passionvine mealybug	Mealybug	Mealybugs have piercing-sucking mouthparts. *P. minor* is a phloem feeder, and this may cause reduced yield, reduced plant or fruit quality; stunting, wilting, discoloration, and defoliation. Indirect or secondary damage is caused by sooty mold growth on honeydew produced by the mealybug. Associated organisms: Ants may be attracted by the honeydew.	Preferred Method. Visual survey of plant material for presence of mealybugs (immatures and adults).	No trap available. No pheromones have yet been identified for *P. minor*.	Infestations can be observed any time during the growing season.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Scirtothrips dorsalis – chilli thrips	Thrips	Damage is caused by sucking out sap from individual epidermal cells leading to necrosis of the tissue. Damage is most severe at the growing tips, on young leaves and shoots, or on flowers and young fruits. *S. dorsalis* rarely feeds on mature leaves. Infested plants become stunted or dwarfed, and leaves with petioles detach from the stem, causing defoliation in some plants. Heavy feeding damage turns tender leaves, buds, and fruits bronze to black in color. Damaged leaves curl upward and appear distorted. On grape, the fruits and leaves are attacked, which causes damage and/or scarring on the leaves or the surface of the berries and on the rachis. The infestation can last until fruit maturation and facilitate the secondary infestation of the fruit by certain flies and fungi.	Preferred Method. *S. dorsalis* is found on the leaves, flowers, and fruits of the hosts. At the initial stage of infestation, the lower surfaces of the leaves become shiny. On individual plants inspect shoots, leaves, flowers, and young fruits for: larvae and adults, and eggs laid inside soft tissue. Particular attention should be given to malformed fruits and foliage (especially foliage at the top of plants). In Florida, 5 to 20 leaves from symptomatic plants are collected at random and placed in a ziplock bag to prevent the adults from escaping. At the lab, foliage is washed in 70% ethanol to remove the adults and immatures. Thrips may be also collected by tapping the plant parts to dislodge thrips. These methods are preferred as they preserve specimens for morphological identification.	No trap available. Not recommended. Sticky traps and Berlese funnels have been used to detect the presence of thrips.	The abundance of chilli thrips is low in the rainy season, but becomes high during the dry season.

Pest	Type	Symptoms	Visual Survey	Traps	Time of Year
Spodoptera littoralis- Egyptian cotton leafworm	Moth	Symptoms include: 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, and fruit. Frass present, often protruding from bore holes. On grape, larvae gnaw holes in the leaves until sometimes only the veins remain. The damage caused by larvae to grapevines is not merely temporary; vines may suffer so severely from exposure to intense sunlight during the summer that their development in the following year will be retarded. Larvae also gnaw at grape bunch stalks, which as a result, dry up, and the larvae feed on the grape berries.	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 is 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 with 2 mg of pheromone blend are replaced after 4 weeks of use. A lure is available from the CPHST- Otis lab. 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. Grape: Larvae scrape the leaf tissue and cause 'drying of the leaves' .The larvae damage the growing berries and cause defoliation. Later instar larvae cut the rachis of grape bunches and petioles of individual berries during the night hours leading to fruit drop.	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	Visual Survey	Traps	Time of Year
Thaumatotibia leucotreta – false codling moth (FCM)	Moth	In general, the habit of internal feeding by FCM larvae displays few symptoms. Fresh larval penetration holes in grapes can be seen, but require careful. Inspection of the fruit. Sometimes a few granules of frass can be found around a fresh penetration hole or a mass of frass may be found around older penetration holes. Sometimes, however, frass is not visible. The area around the penetration hole can become sunken and brown as damaged tissue decays.	Alternative method. Visual inspections of plant materials may be used to detect eggs, larvae, and adults of *T. leucotreta*. Look for plants showing signs of poor growth or rot; holes in fruit; adults hidden in foliage; and crawling larvae. Eggs will commonly be found on fruits, foliage, and occasionally on branches. However, eggs are small and laid singly, which makes them difficult to detect. Fruit should be inspected for spots, mold, or shrunken areas with 1 mm exit holes in the center. On citrus fruits and other fleshy hosts, dissections are needed to detect larvae; larvae are likely to be found in the pulp	Male *T. leucotreta* are attracted to a two component blend of (E)-8-dodecenyl acetate and (Z)-8-dodecenyl acetate. The lure is available from the CPHST- Otis lab. Pheromone lures with (E)- and (Z)-8-dodecenyl acetate may also attract *Cydia cupressana* (native), *Hyperstrotia spp*., Cydia *atlantica* (exotic), *Cydia phaulomorpha* (exotic) and *Cryptophlebia peltastica* (exotic).	Surveys are best conducted during warm, wet weather when the population of the pest increases.

Abbreviated Survey Table

Diseases, Nematodes

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Candidatus Phytoplasma australiense – Australian yellows phytoplasma	Phytoplasma	Symptoms include: irregular chlorosis or yellowing of leaves, which is seen as reddening in red varieties. The chlorotic patches on affected leaves may become necrotic. Leaves of affected shoots can overlap one another. Inflorescences/bunches may die. Berries of more developed bunches may shrivel and fail to ripen. Stems of affected shoots often take on a bluish hue. Later in the season, affected shoots tend to be green and rubbery.	Preferred Method. Visual inspection for symptoms associated with the phytoplasma. Several of the known symptoms should be found together before suspecting a phytoplasma infection on grape. A PCR- based technique can be used to detect the pathogen.	The only known vector of *Candidatus* Phytoplasma australiense is *Oliarus alkinsoni* (not known in the US). This vector is associated with phormium yellow leaf, also caused by *Ca.* Phytoplasma australiense. Other vectors are suspected but not currently confirmed.	Symptoms begin to appear in late spring and increase in incidence until January/February. Infected grapevines are less likely to show symptoms in summer than winter

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Meloidogyne mali – apple root knot nematode	Nematode	Above ground, new primary shoot growth decreases in number and length. Annual gain in plant height, trunk thickness, and numbers of leaves are reduced. Secondary shoots increase in numbers and length. The above ground symptoms are not specific for *M. mali* and may be caused by other nematodes or organisms damaging the roots. Below ground, *M. mali* produces characteristic root-knot nematode galls on roots of host plants. All stages are found associated with the root galls. Eggs, infective second-stage juveniles, and males can also be found in the soil.	Preferred Method. Soil samples must be collected and analyzed by a taxonomic expert. In apple orchards, *M. mali* is most abundant in the top 25 cm of soil. A few nematodes occur 50 cm deep. Horizontally, the nematodes are most abundant in two zones, 20-40 cm and 120-160 cm around the tree. These patterns of nematode distribution are thought to be related to the development and distribution of apple roots in the soil	This nematode is not a known vector.	Infestations can be observed any time during the growing season. In general, sampling for grape nematodes, including other *Meloidogyne* spp., is from Jan.-Feb. Include some feeder roots in the sample.
Monilinia fructigena – brown rot	Fungus	Causes a brown rot primarily on apples, pears, and other pome fruit. Conidial pustules often in concentric circles will be evident within the rotted area. Twigs are rarely infected. Eventually the whole fruit becomes discolored and water is lost so that a mummified fruit is formed. Flowers turn brown, wilt, and collapse.	Preferred Method. Visual inspection for symptoms associated with the fungus. In the past, brown rot fungi were distinguished strictly on cultural and morphological characters. A multiplex-PCR technique can now be used to detect the pathogen and to distinguish it from *M. laxa* and *M. fructicola* (common in US), and *Monilia polystroma* on naturally infected fruit.	Almost any insect has the potential to pick up and carry spores from sporulating mycelium to healthy, susceptible tissues. Birds, wasps (*Vespula* spp.), beetles (*Carpophilus* spp.), flies including *Drosphila* spp. and some Lepidoptera are important in introducing the fungus to healthy, susceptible tissue.	Symptoms appear primarily on mature, ripening fruit; therefore, surveys should coincide with fruiting.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Phellinus noxious – brown root rot	Fungus	Symptoms include: slow plant growth, yellowing and wilting of leaves, defoliation, branch dieback, and plant death Signs of the pathogen, unlike the symptoms, are distinctive for this disease. *P. noxius* forms a thick, dark brown to black crust of mycelium around infected roots and lower stems. The leading edge of the crust is creamy white, glistens with drops of clear, brownish exudate. Patches of white mycelium are present between the bark and sapwood. White, soft, crumbly wood becomes laced with reddish strands of fungus hyphae that turn black with age. Basidiocarps, or fruiting bodies, are purplish brown with yellow-white growing margins and concentric blackish zones towards the edges.	<u>Preferred Method:</u> Visual survey: dark brown mycelial mat or sleeve on the surface of the roots and up to the base of the stem is used reliably for field identification of *P. noxius*. Soil is scraped away around the collar and the main roots and the distinctive mycelial sleeve is often present. Particular attention should be paid to trees that appear wilted or dead. <u>Alternative Method:</u> Baiting out the pathogen by placing sticks of a susceptible host in the soil and retrieving for laboratory examination after 3 weeks is also conducted. The area around the root collar can be mulched for 3 weeks to provide a damp zone that allows the superficial mycelium to progress from the roots onto the trunk of rubber trees. When the mulch is removed, the mycelial filaments of the pathogen can be observed.	No known vector or associated insects	Infestations can be observed any time of the year.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Pseudopezicula tracheiphila – red fire disease	Fungus	Symptoms are lesions on leaves, which are initially yellow on white-fruited cultivars or bright red to reddish brown on red- and black-fruited (*V. vinifera*) cultivars. Later, a reddish brown necrosis develops in the center of the lesion, leaving only a thin margin of yellow or red tissue between the necrotic and red areas of the leaf. The lesions are typically confined by the major veins and the edge of the leaf and may be several centimeters wide. Early infections generally result in less loss than late infections. Inflorescences can be attacked before or during flowering, causing them to rot and dry out.	Preferred Method. Visual inspection for symptoms associated with the fungus. Identification is dependent upon observation of cultural and morphological characters of the fungus.	No known vector or associated insects.	Initial infections may occur on the first to the sixth leaves of young shoots, resulting in minor losses. Later infections may attack leaves up to the 10th or 12th position on the shoot and can be result in severe defoliation. In addition, the fungus may attack inflorescences before or during bloom.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Xiphinema italiae – dagger nematode	Nematode	Feeds along feeder roots causing root discoloration and stunting.	Preferred Method. Soil samples must be collected and analyzed by a taxonomic expert. Samples in the viticulture region of Greece were taken using a soil auger to a depth of 30 cm. Soil samples were put in plastic bags, transported to a diagnostic lab, and extracted using a modified decanting and sieving method with a final separation from fine soil particles after migration through a 95 µm filter.	This nematode has been shown to be a vector of *Grapevine fanleaf virus* (GFLV).	Infestations can be observed any time during the growing season. In general, sampling for grape nematodes, including other *Xiphinema* spp., is from Jan.-Feb. Include some feeder roots in the sample.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Xylella fastodiosa CVC strain – citrus variegated chlorosis	Bacterium	Symptoms include: include a foliar interveinal chlorosis (yellowing) resembling zinc deficiency on the upper surface of young of leaves, small light-brown gummy lesions develop on the lower surface of these leaves as they mature., The leaves are often smaller than normal. Infected trees show stunting and reduced growth rates, with twig and branch dieback and canopy thinning. Blossom and fruit set occur at the normal time, but the usual fruit thinning does not occur, resulting in clusters of 4 to 10 early-maturing small fruit. Although sweet orange trees affected by the disease do not die, fruit from the trees may be severely undersized, have hard rinds, lack juice, acid-flavored, and are of no commercial value. The sugar content of the fruit is higher than in non-affected fruit, and the fruit ripen earlier than normal.	Preferred Method. Survey for CVC will involve a visual inspection of citrus and/or grape for characteristic symptoms. While CVC may occur on all cultivars, species, and hybrids of citrus, symptoms vary depending on the particular plant and its age. Sweet oranges are the most susceptible and show the most severe symptoms. Grapefruit, limes, mandarins, and mandarin hybrids show less severe symptoms. Rangpur lime, lemons, citron and pummelo are tolerant to the disease, showing only very mild (if any) symptoms. Young infected trees generally show more severe foliar symptoms than mature infected trees.	In Brazil, 12 of 16 sharpshooter species tested were able to transmit the bacterium. In the United States, the blue-winged sharpshooter (*Oncometopia nigricans*) and the glassy winged sharpshooter (*Homalodisca coagulata*) have been shown experimentally to be a vector of the CVC strain of *X. fastidiosa*.	Symptoms can be observed at any time during the growing season.

Pest	Type	Symptoms	Survey Methods	Vectors / Assoc. Insects	Time of year
Xylophilus ampelinus – bacterial blight of grapevine	Bacterium	The bacteria attack the vascular tissues, causing shoot blight, canker, and occasionally leaf spots. In early spring, bud break on affected spurs is either delayed or does not occur. Other spurs on the same vine grown normally. Bud break may occur on only a few spurs, and some of the developing shoots may be stunted, weak, and/or chlorotic, with dark brown streaks on one side. These shoots will eventually wilt and die. During this period, affected branches and spurs appear slightly swollen because of hyperplasia of the cambial tissues, which have a soft, cheesy consistency. Longitudinal cracks occur in the bark at the swollen regions.	Preferred Method. Visual inspection is conducted by observing symptoms associated with the bacteria. Standard bacteriological tests can be used, but indirect immunofluorescence technique and sensitivity to phage φ 15 will allow rapid identification. A PCR technique has also been described.	No known vector or associated insects.	Symptoms are most conspicuous in early spring until midsummer. The first symptoms on the young, tender shoots appear two or three weeks after the buds begin to grow.

Appendix A: Diagnostic Resource Contacts

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Appendix B: Glossary of Terms

Aedeagus: In male insects, the penis or intromittent organ, situated below the scaphium and enclosed in a sheath.

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

Agglomerate: To form or collect into a rounded mass.

Allopatric: Occurring in separate, non-overlapping geographic areas. Often used to describe populations of related organisms unable to crossbreed because of geographic separation.

Androconia (also called scent scales): are modified wing scales on butterflies and moths that release pheromones. Only males have these scent scales.

Apophysis: A natural swelling or enlargement found at the base of the stalk or seta in certain mosses or on the cone scale of certain conifers.

Areolated: Divided into small spaces or areolations; usually pertains to the cuticle of a nematode.

Arthrospore: A fungal spore resulting from the fragmentation of a hypha.

Bolls: The spherical shaped fruits of cotton and flax.

Calyx: The outer-most group of leaves surrounding the flower; the external-most part of the flower.

Cerarii: These are characteristic of mealybugs and consist of groups of large setae, usually conical, on the lateral margins of the body.

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

Chlorotic: Abnormal condition of plants in which the green tissue loses its color or turns yellow as a result of decreased chlorophyll production due to disease or lack of light.

Cisanal setae: In coccids, the shorter and further two of the four setae (commonly known as hairs) near the caudal ring.

Clamp connection: A bridge- or buckle-hyphal protrusion in basidiomycetous fungi, formed at cell division and connecting the newly divided cells.

Cockchafer: Any of various large European beetles destructive to vegetation as both larvae and adult.

Coleoptile: A protective sheath enclosing the shoot tip and embryonic leaves.

Cornuti: Literally, the horned ones, which was the name given to a Byzantine auxilium palatinum.

Corona: A crown; the whorl of structures between the corolla and stamens.

Costa: Any elevated ridge that is rounded at its crest; the thickened anterior margin of any wing, but usually the forewings of an insect.

Coxae (pl. of coxa): The basal segment of the leg of an insect, by means of which it is articulated to the body.

Cremaster: 1) The apex of the last segment of the abdomen; 2) the terminal spine or hooked process of the abdomen of subterranean pupa, which is used to facilitate emergence from the earth; 3) an anal hook by which some pupae are suspended.

Deutonymph: The third instar of a mite.

Diapause: A period of arrested development and reduced metabolic rate, during which growth, differentiation, and metamorphosis cease; a period of dormancy not immediately referable to adverse environmental conditions.

Digitus: Having appendages of the feet (as found in member of the family Coccidae), which may be either broadly dilated or knobbed hairs; tenent hairs, empodial hairs.

Discal: On or relating to the disc of any surface or structure.

Ecdysis: Molting; the process of shedding the exoskeleton.

Endocarp: The hard inner layer of the pericarp of some fruits that contains the seed.

Epiphytotic: Epidemic among plants of a single kind, especially over a wide area.

Exuvia: The cast skin of an arthropod.

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

Filiform: Thread-like or hair-like.

Flaccid: lacking in strength or firmness or resilience.

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

Gubernaculum: Nematodes: Spicule guide; sclerotized accessory piece.

Hemispherical: Shaped like the half of a globe or sphere.

Hemizonid: Nematodes: Lens-like structure situated between the cuticle and hypodermal layer on the ventral side of the body just anterior to the excretory pore; generally believed to be associated with the nervous system.

Hyaline: Like glass, transparent and colorless.

Incipient: Beginning to exist; coming into existence.

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

Leeward: On the side away from the wind.

Lodging: To fall over.

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

Lychees: The fruit of a tree native to China. It is nutlike, having a rough but tender shell and containing an aromatic pulp and a single large seed.

Metanotum: The upper surface of the third (posterior) thoracic segment (metathorax).

Micropile: A very small opening in the outer coat of an ovule, through which the pollen tube penetrates; the corresponding opening in the developed seed; one of the minute openings in the insect egg, through which spermatozoa enter in fertilization.

Microtrichium (pl. microtrichia): Small, sclerotized, and non-innervated cuticular projects on the body and wings of insects; which are also found on the tracheae.

Monandrous: Having only one male mate at a time.

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

Neonate: A recently born larva.

Ocelli: A simple eye of an insect or other arthropod.

Orbicular: Circular in outline.

Ostioles: In Heteroptera, one of the lateral metasternal external openings of the stink gland, placed near the coxa in the adult, but paired and dorsal on the abdomen of the nymph. For plant pathology, pore; opening in the papilla or neck of a perithecium, pseudothecium, or pycnidium through which spores are released

Oviparous: Reproduction in which young hatch from eggs.

Palearctic: The biogeographic region including Europe, Asia north of the Himalayas, and Africa north of the Sahara.

Parthenogenetic: Capable of reproduction without mating or male fertilization.

Parthenogenesis: Process of reproduction by the development of an unfertilized egg.

Pedicel: Small slender stalk; stalk bearing an individual flower, inflorescence, or spore.

Phasmids: Any of various stick-like or leaf-like insects, including walking sticks and leaf insects.

Phenology: The relationship between the climate and biological events, such as flowering or leafing out in plants.

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.

Phytophagous: Plant-eating.

Pinaculum: In caterpillars, an enlarged seta-bearing papilla forming a flat plate.

Pleural membrane: A thin serous membrane in mammals that envelopes each lung and folds back to make a lining for the chest cavity.

Polygynic: Phenotypic trait whose expression is controlled by, or associated with, more than one gene.

Polyphagous: Eating many kinds of food.

Pome fruits: A fleshy fruit, such as an apple, pear, or quince, having several seed chambers and an outer fleshy part largely derived from the hypanthium. Also called false fruit.

Prolegs: 1) Any process or appendage that serves the purpose of a leg; 2) specifically, the pliant, non-segmental abdominal legs of caterpillars and some sawfly larvae. Not true segmented appendages.

Pronotum: The upper (dorsal) plate of the prothorax.

Protonymph: Second instar of a mite.

Pygidium: Pertaining to the pygidium (*i.e.*, the last dorsal segment of the abdomen).

Quiescent: Quiet and at rest, but not necessarily dormant and having the potential for resumed activity; can apply to non-meristematic cells.

Rachis: Floral thorn or point.

Reniform: Kidney-shaped.

Reticulate: Descriptive of surface sculpture, usually the insect's integument, which is covered with net-like lines.

Saccus: A sac.

Sclerotized: Hardened.

Senescence: The last stage in the post-embryonic development of multicellular organisms, during which loss of functions and degradation of biological components occur. A physiological ageing process in which cells and tissues deteriorate and finally die.

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

Setae: Bristles; commonly known as hairs.

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

Spatulate: Rounded and broad at the top, attenuate at base. Shaped like a spoon, with a narrow end at the base.

Spiracles: Breathing pores; in the plural the lateral openings on the segments of the insect body through which air enters the trachea.

Sternites: The ventral shield or plate of each segment of the body of an insect or other arthropod.

Striated: Numerous parallel, fine, and impressed lines.

Stylet: A stiff, slender, hollow feeding organ of plant-parasitic nematodes or sap-sucking insects, such as aphids or leafhoppers.

Symptom: Indication of disease by reaction of the host, e.g. canker, leaf spot, wilt (see sign).

Tarsus: The leg segment immediately beyond the tibia, consisting of one or more segments or subdivisions.

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

Tergite: A dorsal sclerite or part of a segment, especially when such part consists of a single sclerite.

Termen: The outer margin of a wing, between the apex and the posterior or anal angle.

Tillers (subterranean): A lateral shoot, culm, or stalk arising from a crown bud; common in grasses.

Univoltine: Having only one generation per year.

Variegated: Varied in color, of several colors in indefinite patterns.

Vesica: Lepidoptera: the penis, or terminal part of the aedeagus. The vesica is membranous and eversible, typically held within the tubular part of the aedeagus, but everted and inflated during copulatation.

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 broadwayi*																																		0
118	*Coconut cadang-cadang viroid* (CCCVd)	Coconut Cadang-Cadang Disease																																	0
119	*Acroceras zizanioides*	Oat Grass												■																	■	■	■	■	5
120	*Rubus alceifolius*	Giant Bramble																													■	■	■	■	4

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				■		▲									■	■				■					■	■		▲	■				9
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			■		■			▲			■				■				■		▲				■		■	■					10
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

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**

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**	**12**	**20**	**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.