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Cooperative Extension
Agricultural Experiment Station
Statewide IPM Project

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IN THIS ISSUE

ARTICLES

BIONOMICS OF THE OLIVE FRUIT FLY *BACTROCERA (DACUS) OLEAE*

Richard E. Rice, U.C. Kearney Agricultural Center

The olive fruit fly (olive fly) *Bactrocera oleae* (Gmelin) (Diptera: Tephritidae) is the most serious insect pest of olive fruit in the world. It is known primarily from the Mediterranean area of southern Europe, and is also found in North Africa, the Middle East, and along the east coast of Africa to South Africa (Table 1). There are records of olive fly infestations in fruit three centuries B.C. in the eastern Mediterranean area. It is generally agreed among olive fly researchers that this insect can survive and develop in any area of the world where olive trees are grown.

Distribution of olive fly in California

The olive fly was first detected in California on October 19, 1998 in a McPhail trap placed in an orange tree in west Los Angeles, CA. This trap was part of the monitoring and detection trapping program for Mexican fruit fly and other fruit flies that do not respond to specific lures. Following the initial detection, delimitation trapping showed all of the coastal counties from Santa Barbara south to San Diego, and inland to Riverside and San Bernardino counties to be generally infested with olive fly. Trapping in the coastal counties was terminated in early 1999 due to the widespread infestations present in these areas.

Table 1. World distribution of the Olive Fly, *Bactrocera oleae*.

Albania	Israel	Sardinia
Algeria	Italy	South Africa
Canary Islands	Jordan	Spain
Corsica	Lebanon	Syria
Cyprus	Libya	Tunisia
Egypt	Mexico	Turkey
Eritrea	Morocco	United States
France	Pakistan	Yugoslavia
Greece	Portugal	

The first olive fly in the San Joaquin Valley was trapped in a commercial grove on September 14, 1999 near Plainview in southern Tulare County. Intensive trapping around this single fly find through the remainder of 1999 failed to detect any additional flies in this area, or elsewhere in the San Joaquin Valley. However, on May 5, 2000 a sexually mature female fly was trapped in the Mayfair area east of Bakersfield, Kern County. As with the initial find in west Los Angeles, this fly was trapped in an orange tree in a McPhail trap. A second fly was trapped near Bakersfield on May 16, 2000, this time a male in a ChamP yellow panel sticky trap specific for olive flies. Another olive fly was detected in Reedley, Fresno County, on May 17, 2000. On May 22, 2000 the first olive fly on the California coast north of Santa Barbara County was trapped in Arroyo Grande, San Luis Obispo County. A second fly was trapped in Tulare County on May 23, 2000 near Terra Bella. Following these additional detections in the San Joaquin Valley and in San Luis Obispo County, olive fly collections increased rapidly in all of these areas. Numbers of olive flies collected in counties north of Los Angeles are shown from July 12, 1999 through July 10, 2000 (Table 2).

Table 2. Collections of Olive Fly in California north of Los Angeles County[1/].

County	First Collection		Trap[2/]	Host	No. Flies		Trapping Terminated
					Males	Females	
Ventura	7/12/99	F	Ch.	Olive	71	36	3/9/00
Santa Barbara	7/23/99	M	Ch.	Olive	122	90	12/11/99
San Luis Obispo	5/22/00	M	Ch.	Olive	72	17	
Kern	5/5/00	F	McPh.	orange	22	8	
Tulare	9/14/99	M	Ch.	Olive	143	34	
Fresno	5/17/00	M	Ch.	Olive	9	5	
Madera	6/19/00	M	McPh.	Loquat	3	0	
					412	183	

[1/] Through July 10, 2000.
[2/] Ch. = ChamP; McPh. = McPhail.

Survey and detection trapping in Ventura and Santa Barbara counties has now terminated due to the generally invested nature of those two areas. Detection trapping is continuing in San Luis Obispo County although fly numbers are increasing rapidly in that area as well, primarily in urban ornamental trees. The first fly detection in Madera County was noted on June 19, 2000. As of the publication date of this issue of the UC PPQ however, numbers shown in Table 2 are already out of date because of the rapidly increasing detections of flies throughout the southern San Joaquin Valley. (Addendum: On July 5 and July 7, singe male olive flies were trapped in Fremont, Alameda County, and San Jose, Santa Clara County, respectively.) Two additional flies were trapped in Alameda County on July 19, 2000.

In addition to the expanding infested areas of California, detection trapping in the summer and fall of 1999 in Baja California, Mexico showed the presence of olive fly as far south as the San Vicente region of western Baja.

Description

The adult olive fly is normally 4-5 mm long with large reddish eyes and small antennae. The thorax is dark brown with 2-4 gray or black longitudinal stripes. The scutellum is yellow to white; there are also several yellow-white patches on each side of the thorax. The abdomen is brown with darker areas on the sides of each segment (this character is quite variable). The wings of olive fly are clear except for a small distinct black spot at the tips; wing veins may also be slightly dark. Olive fly does not have colored wing bands or patterns typical of many other species of fruit flies such as the Mediterranean fruit fly, *Ceratitis capitata*.

Figure 1. Adult female olive fly, *Bactrocera oleae.*

Hosts of olive fruit fly

Fruits in the plant genus *Olea* are the only known natural hosts for *B. oleae.* Many cultivars of edible olives (*O. europea*) can be infested. In general the larger table olive varieties are preferred for oviposition by female flies; these fruits tend to allow better survival and produce greater numbers of olive fly larvae. However, even the smaller oil olive cultivars are excellent hosts for olive fly wherever these cultivars are grown.

In addition to cultivated olives, olive fly is known to attack several species of wild olives. Infestation in these hosts has allowed the fly to spread along the east coast of Africa as far as central South Africa where wild olives occur along with a few plantings of commercial olives. As might be suspected from such a narrow host range, olive fly has very specific and restrictive nutritional requirements. It has been shown that *Pseudomonas savastanoi,* the bacterial causal agent of olive knot disease, is a symbiont required in the gut of olive fly larvae and adults. The bacteria help flies break down chemicals in olive fruit into essential amino acids and proteins required for growth and reproduction. Olive flies can be reared on artificial diets, but with greater difficulty than many other tephritids.

Although *O. europea* or other species of olives are the only natural breeding host for olive fly, flies have been trapped in other plants where they search for food, or for protection and refuge. In addition to olive, adult fly collection hosts in California include orange, grapefruit, tangerine, calamondin, cherry, plum, lemon, avocado, loquat, nectarine, *Myoporum*, and Surinam cherry. Flies trapped in these nonbreeding hosts are often caught in McPhail traps, rather than in the yellow ChamP traps that are normally placed specifically in olives. Trapping and migration studies from Crete list additional nonbreeding hosts such as walnut, apple, sycamore, chestnut, vines, fig, *Arbutus*, and persimmon. Tomato and *Ligustrum* (privet) are reported as laboratory hosts supporting olive fly larval development.

As indicated earlier, olive cultivars in Mediterranean countries show varying susceptibility to infestation by olive fly. In general, larger sizes of olives and olives with higher water content (e.g. table cultivars) are more susceptible than small olives with lower water content (oil cultivars). In Greece, the smaller oil cultivar Koroneiki is less susceptible to olive fly infestation than is the larger table olive cultivar Tsounati.

Olive fly phenology and biology

Temperature relationships and developmental thresholds for olive fly are similar to other tephritid fruit fly species (Table 3). In California it is believed that at least three and perhaps as many as five or six generations of olive fly can develop in the olive production areas of the San Joaquin and Sacramento valleys. It appears that in mild southern areas of the state such as San Diego County, olive fly development can be continuous throughout the year. Olive fruit remaining on trees through the winter into early summer and presence of mated female flies contribute to this development.

Table 3. Developmental thresholds for *Bactrocera oleae* at constant temperatures.

Stage	Lower	Upper
Egg	43º – 46º F	95º – 100º F
Larva	39º – 46º F	95º F
Pupa	41º – 48º F	86º F
Adult	40º F	102º F

The adult activity threshold for olive fly is approximately 15.5º C (60º F). In summer olive flies can complete a generation in as little as 30 to 35 days at optimum temperatures. The eggs hatch in 2 to 3 days and larvae will develop in approximately 20 days during the summer and fall. Pupal development requires approximately 8 to 10 days during the summer but may last for six months in winter. Unlike other tephritid species, olive flies pupate within the host fruit during warmer months, but leave the fruit to pupate in the ground or in any protected niche during winter. Adult flies can live from 2 to 6 months depending upon food (honey dew, bird feces) availability and temperatures of their environment. Male flies are polygamous; females are normally monogamous. Some authors report olive fly females laying from 200 to 500 or more eggs, thus the reproductive potential for olive flies is extremely high if host fruit is available for oviposition.

According to European literature, olive flies survive best in more humid coastal climates but are also known to heavily infest olives that are grown in interior dry regions of Greece, Italy, and Spain. One report from 1929 describes infestations approaching 100% in olives from Cordoba and Jaen, Spain. High temperatures in the 38-41º C (100-105º F) range are detrimental to adult flies and to immature stages in fruit. However, adult flies are very mobile and have the ability to seek out more humid areas within olive groves, particularly those that are heavily irrigated, or in more humid urban environments. Various authors have reported adult fly movements from 200 m (656 ft) in the presence of olive hosts, to as much as 4000 m (2.5 mi) to find hosts.

Dispersals up to 10 km (6.2 mi) have been reported over open water in the Mediterranean. Presence or absence of fruit modulates fly dispersal in groves.

The seasonal phenology of olive fly varies considerably depending upon the area and climate of the world that it is infesting. In general, olive flies overwinter as pupae or in the adult stage. Females can lay eggs in fruit in warmer climates throughout the winter. Overwintered adult populations decline to generally low levels by February or March, however new adults from overwintered larvae and pupae begin to emerge in March and April in many Mediterranean climates (Fig. 2). These adult populations then also begin to decline during May and June. The next generation of adults appears in July and August at the time that new crop olive fruits become susceptible to infestation. Fruit susceptibility begins at the time of pit hardening, usually in July in the Mediterranean areas. In California, however, pit hardening was observed in olives in Tulare County (along with many trapped flies) in early June 2000. Mature female flies were being collected in mid June; mature eggs in mated females were present in late June.

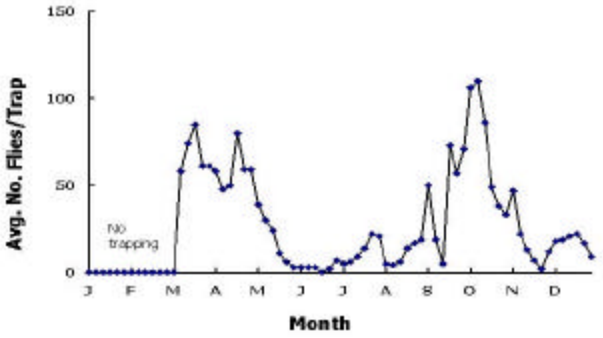


Figure 2. Phenology of the Olive Fly, *Bactrocera oleae*, Mochos, Crete, 1981. Haniotakis et al., 1986.

Additional generations of flies are produced through the late summer and fall months and into December, depending upon fruit maturity and availability on the trees. Olives that are left on trees can produce high numbers of flies from late fall to early spring if these fruits are unharvested, or are allowed to simply mature and drop naturally from the trees before being collected for oil pressing. There will undoubtedly be significant differences in the seasonal phenology and biology of olive fly in California due to the numerous microclimates found within our coastal and interior regions.

Economic impact

The economic impact and monetary losses due to infestations by olive fly vary considerably and depend primarily on the end use of the fruit. Economic thresholds for *B. olea* in table olives are extremely low. In many countries infestation levels of 1% or less are required for high quality production of table olives. This would certainly be true for California growers. Oviposition stings alone, without eggs or larval feeding, will lower the value of table fruit. Oil cultivars can sustain higher infestations as long as the fruit is harvested within a relatively short period of time (3 to 4 weeks) after the larvae begin feeding in the fruit. A decline in oil quality occurs due to secondary infestations of bacteria and fungi that greatly increase the acidity of the oil. European authors have indicated economic losses of table olive crops as high as 100% from infestations that are not controlled. Oil losses can range as high as 80% from combined fruit drop, pulp destruction, and increased acidity of oil if fruit is not harvested in a timely fashion. A general example of the variation in olive fly impact on table olive and oil production in the Mediterranean area is shown in Table 4, primarily for olive oil cultivars. The potential for extreme economic losses to the California olive industry is great because the majority of our olives are produced for table consumption rather than for oil production.

Table 4. Economic impact of the Olive Fly, *Bactrocera oleae*, (olive production loss).[1/]

Spain	1962	5%
Italy	1962	25%
Greece	1962	30-35%
Israel	1962	20-60%
Cyprus	1962	15-20%
Yugoslavia	1962	20-40%
Libya	1980	27-72% (avg. 50%)
	1981	7-14%
Portugal	1994-96	4-8%, table; 17-19%, oil

[1/] Primarily oil cultivars.

Quarantines and control

Management of *Bactrocera oleae* in California will depend on a combination of early detection and delimitation trapping, limited interior quarantines, effective bait sprays or attract and kill trapping of adult flies, cultural practices (e.g. timely harvest), and fruit sanitation. As of July 2000 specific details on olive fly management in production areas are still being developed. In general, however, control of olive fly for the immediate future will rely upon protein hydrolysate bait sprays containing spinosad insecticide, and perhaps a mass trapping or attract and kill strategy based on insecticide-treated traps containing both ammonium bicarbonate and pheromone (spiroketal) lures. Restrictions and transporting requirements will also be placed on movement of harvested fruit from infested San Joaquin Valley counties to the processing facilities in

northern California where olives are also grown. In addition, there are plans for mass trapping in olive trees within ¼ mile of known infested commercial groves. Olive trees in urban or noncultivated areas near commercial groves will be trapped in an attempt to reduce fly populations and migrations of adult flies back to production groves.

Prognosis for olive fly in California

Although there is a tremendous amount of literature from Mediterranean researchers on olive fly biology and control, the ultimate impact of olive fly and management of this pest in California is still undetermined. Although our climate is very similar in many respects to areas of the world that grow olives and are infested with olive fly, we will still need to develop our own specific phenology and biology data for this pest. Hopefully this will lead to development of IPM programs for olives that will allow California growers to continue economical production of high quality table and oil olives while at the same time not disrupting the current very effective IPM programs for other olive pests such as scale insects.

GLASSYWINGED SHARPSHOOTER MOVES INTO THE SAN JOAQUIN VALLEY

Walt Bentley, U.C. Kearney Agricultural Center

The glassywinged sharpshooter, *Homalodisca coagulata* Say, is rapidly moving into the San Joaquin Valley. The initial find in the Valley was made in fall 1998 in an organically managed table grape vineyard. The vineyard is located just east of Bakersfield near the town of Edison. Edison is one of the earliest citrus producing areas in the state and, unfortunately, the glassywinged sharpshooter (GWSS) survives well on citrus.

Since that first San Joaquin Valley identification, GWSS has now been found in Porterville, Traver, North and south Fresno, Kingsburg, Madera, Lodi, and Sacramento. Currently there have been no positive finds in Merced or Stanislaus counties. Most of these findings have been in residential areas; the most abundant populations being in Porterville and the Sunnyside area of Fresno. Commercial vineyard infestation is occurring in Kern County and the insect is widespread in the city limits of Bakersfield.

Most of the focus to this point has been on identifying the distribution of GWSS, which carries the bacterium *Xylella fastidiosa*, the causal agent of Pierce's disease. Residential spray programs have been undertaken in Porterville and Fresno. Carbaryl, a carbamate registered for ornamental, vegetable, and fruit applications, has been sprayed to reduce the GWSS populations in these areas. The treatments have been made under the supervision of the Fresno and Tulare County Ag Commissioners and strict precautions were followed to reduce drift. Initial kill of the exposed sharpshooters has been good, but residual control is short. Live sharpshooters have been found in sprayed areas a week after treatment. In general, the public has reacted quite positively to these treatments.

In addition to delineating the spread of GWSS, the distribution of *X. fastidiosa* must now be determined. We know that there are well over 60 plant species commonly found in the San Joaquin Valley on which GWSS can survive. We also know at least 75 plant species that harbor the bacteria. Most of them do not show symptoms of Pierce's Disease. Key plants which are known hosts for the bacteria include wild grape, Himalayan blackberry, California blackberry, hemlock, small periwinkle, elderberry, California bay laurel, California buckeye, Oregon ash, and Valley oak. We can only guess at the number of ornamentals being brought in from other parts of the United States or other countries that also harbor *X. fastidiosa*. Currently, a diagnostic laboratory is being established at UC Davis with funds being provided by state and federal governments. Farm Advisors have been given a sample allocation to use this laboratory in confirming suspected Pierce's disease in grapes, but the facility will be unable to handle the wide range of suspected host plants for the bacteria.

The northern San Joaquin Valley and the north coast have a history of Pierce's Disease and are certainly most vulnerable to spread if GWSS becomes established. In the southern San Joaquin Valley, Tulare County has such a history and is the most vulnerable county. Areas around Traver, Woodlake, Lindsay, and the grape growing area north of Visalia have been plagued for decades by the disease. It is critical that GWSS be kept out of these areas.

For now, both farmers and pest control advisers must be extremely vigilant in identifying both the GWSS and symptoms of Pierce's Disease in the vineyards. The best way to do this is to visit the following websites:

www.CNR.Berkeley.edu/xylella/
http://ucceventura.xlrn.ucsb.edu/IPM/
http://danrcs.ucdavis.edu/Special/gwss/default.shtml

As the season progresses updates on chemical and biological management will be provided at these sites. They will be the sources of the most current information.

PROTECTING VINEYARDS FROM PIERCE'S DISEASE VECTORED BY THE GLASSY-WINGED SHARPSHOOTER: PRELIMINARY OBSERVATIONS

Phil A. Phillips, UCCE Ventura Co.

The first line of defense for vineyards outside the current range of the glassy-winged sharpshooter (GWSS) is avoiding, or at least delaying, a new infestation of this insect vector of bacterial plant pathogens, particularly the bacterium *Xylella fastidiosa* which causes Pierce's Disease (PD). Growers should take every precaution to avoid the introduction of this new bacterial vector into their area. Potential avenues of spread such as the movement of infested nursery stock (all life stages), in harvested grapes (adults), or in vehicles (adults and nymphs) returning from infested areas during the season need to be shut down. Importation of landscape nursery stock from infested nurseries in southern California, Kern County or other known areas of infestation for the purpose of home or office beautification projects should be avoided.

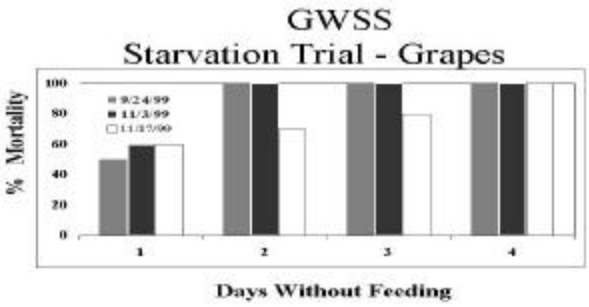
Like native sharpshooters, the glassy-winged sharpshooter (GWSS) will have to be managed outside the vineyard. There are several possibilities which growers are already considering even in the absence of documented success. Border plantings of trap crops such as an early, vigorous grape variety or rootstock (e.g. *Vitis rupestris*) or young lemon trees may provide some protection. GWSS should be attracted to these prior to the vineyard leafing out in the spring. Repeated contact insecticide applications to these trap plants might help prevent successful movement into the vineyard. More innovative solutions may also be warranted, especially when the vineyards are located adjacent to excellent hosts such as citrus. Some Temecula area grape growers have attempted to set up lease agreements for portions of their neighbors' citrus orchards. In this way they could establish control of the GWSS situation within the adjacent citrus plantings. By managing GWSS populations within the adjacent citrus through repeated insecticide applications, they hope to stop the sharpshooters at an important source.

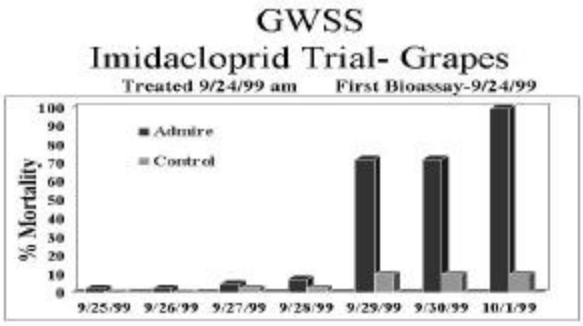
Managing GWSS inside the vineyard will pose a near heroic challenge since this introduced sharpshooter, unlike native sharpshooters, likes grapevines and will breed within vineyards. This insect appears to be relatively easy to kill. Insecticides such as chlorpyrifos (Lorsban), which is typically used for scale insect control in citrus, is just one example. GWSS was suppressed for nearly three months after a chlorpyrifos application for red scale was made in early April of 1999 in a three-year old lemon orchard in Ventura County. Short-lived contact insecticides, even if repeatedly used, will likely leave windows of opportunity for GWSS transmission of *X. fastidiosa.* Long-lived, systemic insecticides such as imidacloprid (Admire, Bayer Corp.) can suppress GWSS populations for at least several weeks and possibly up to two months or more with minimal if any impact on beneficial species. In an experiment conducted in the same three-year old Ventura County lemon orchard during 1999, imidacloprid was applied through the mini-sprinkler irrigation system and resulted in 91-95% reduction in GWSS activity for up to two and a half months. However, evidence from this replicated trial indicated that the sharpshooters were not killed out right by the treatment (as they are with the chlorpyrifos) , but rather avoided the treated trees and aggregated into trees within the untreated plots. Insects caged on treated trees did not die right away and continued to feed, but at a greatly reduced rate compared to those caged on untreated trees and eventually died of starvation after a week or more of being caged.

In a similarly treated vineyard in Ojai during 1999, GWSS were observed to continue to feed on treated vines for one week or more depending upon how soon after treatment they were exposed to the treated vines. Reduced feeding activity was again observed with those insects placed in sleeve cages on treated vines compared to those on the untreated controls. It is not known if feeding times on the treated vines were sufficient for transmission of the *Xylella* bacterium had they been carrying it. Four replicated bioassays were conducted, each at a successively greater interval post-treatment. Exposure at greater intervals post-treatment resulted in longer survival times before death ensued, presumably from starvation/dehydration (Figures 1a-d). Insects held on treated canes all survived longer than insects held in cages within the vine canopies, but without access to food (canes). These control insects all died from starvation/dehydration within the first 48 hours in the bioassays conducted during September and October when maximum daytime temperatures were 90-95^{o} F and within 96 hours for a later bioassay conducted during mid-November when maximum temperatures were 75-80^{o} F.

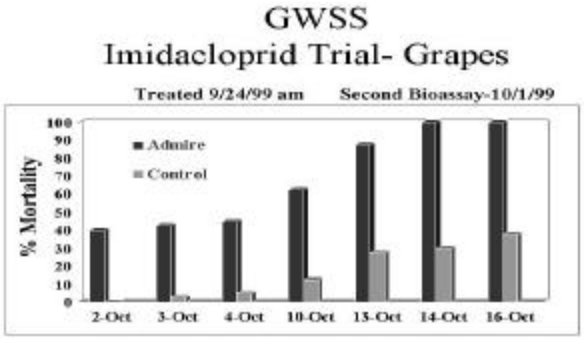
In another research project using annual systemic imidichloprid applications alone from 1995 to 1997,

esearchers at the University of Georgia clearly failed to protect a newly planted vineyard of *vinifera* grapes. They merely achieved an 18-month delay in the eventual 100% infection of the vineyard with PD. In this study, there were additional sharpshooter species involved, but GWSS was the predominant species responsible for considerable vector feeding pressure. Vector pressure along with bacterial inoculum sources from outside the vineyard are critical elements in the success of disease prevention programs within a vineyard. A "within vineyard" treatment strategy for grape growers will likely fail if used alone rather than in combination with other management options outside the vineyard.

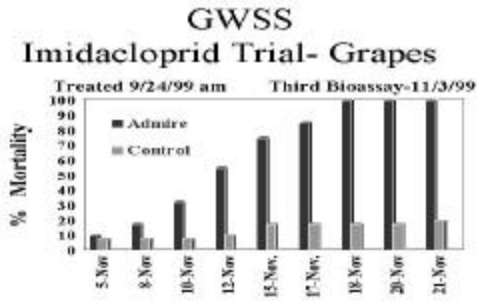




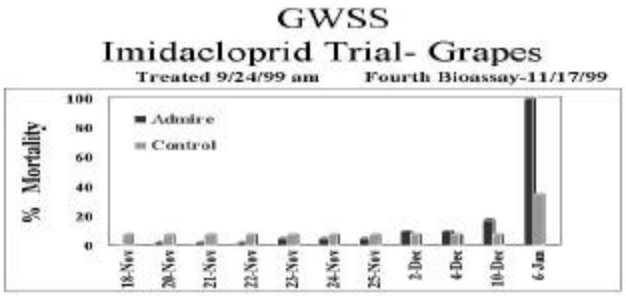
GWSS mortality after continual exposure to Admire-treated vines during the first week post-treatment.



GWSS mortality after continual exposure to Admire-treated vines during the second and third weeks post-treatment.



GWSS mortality after continual exposure to Admire-treated vines during the sixth, seventh and eighth weeks post-treatment.



GWSS mortality after continual exposure to Admire-treated vines during the eighth through the fifteenth week post-treatment.

HAIRY FLEABANE BIOLOGY AND CONTROL

Timothy S. Prather, U.C. Kearney Agricultural Center

Hairy fleabane, *Conyza bonariensis* (L.) Cranq., is an annual weed that has increased in abundance throughout the tree and vine growing regions of the San Joaquin Valley. Hairy fleabane can form dense stands that interfere with proper distribution of water, particularly through low volume sprinkler and drip irrigation systems. It is difficult to control unless applications of preemergent and postemergent herbicides are well timed.

Identification

Hairy fleabane can be confused with a related species called either horseweed or marestail (*C. canadensis*). When plants have cotyledons, the two may be separated. Hairy fleabane cotyledons are longer than they are wide while horseweed cotyledons are nearly as wide as they are long. They are not distinguishable until after the twelve leaf stage if the cotyledons are not present. After the 12 leaf stage the hairy fleabane leaves are much narrower and hairs on the leaves are stiff. There are greater distances between leaves as well. Horseweed tends to have very little main stem visible between leaves. As plants get older, hairy fleabane develops side

branches along the main stem but horseweed does not. Hairy fleabane develops flower heads that are about 3/16 inch across with horseweed flower heads 1/8 inch or less across.

Phenology

Hairy fleabane usually emerges in mid February or early March. However, during warm winters it does emerge earlier. In 1998, hairy fleabane had more than 18 leaves by January 10 at the Kearney Agricultural Center. During warm winters some plants may also survive the entire winter and continue to produce seeds into the spring. However, in 1999 all mature plants that were labeled and watched through the winter died indicating that hairy fleabane usually does not survive the winter. The seedlings emerge in late winter and grow through the spring and summer, remaining green and producing seed all during the summer and into the fall. As the plant develops in early spring through summer, hairs on the leaves becomes more dense making it less susceptible to contact herbicides like paraquat (Gramoxone).

Control Timing

Hairy fleabane is controlled with the herbicide simazine (Princep). It is also controlled with the herbicide thiazopyr (Visor) that currently is registered in citrus. In most years, applications made through the month of February should control hairy fleabane. Warm winters that are the result of El Niño will allow earlier germination and so preemergent applications made in January and February should include an effective postemergent herbicide. Postemergent control must be properly timed to adequately control this species. If the plants are under the 8 leaf stage then 1.6 quarts of Roundup Ultra (or equivalent of Roundup Classic, Touchdown or Glyphos) has controlled at least 90% of the plants (Figure 1). Above 8 leaves, over 2.1 quarts of Roundup Ultra were required (Figure 1). Adequate control is not achieved when plants have 14 or more leaves.

The closely related species, horseweed, seems to germinate later than hairy fleabane. However, the two species may be present at the same time during a postemergent application. Overall, horseweed appears less susceptible to glyphosate (Figure 2) at the younger stages, but increasing the rate of application does increase control. The two species hairy fleabane and horseweed are difficult to distinguish when they are young and susceptible to postemergent herbicides. Usually there are escaped plants that flower, and at this stage, they are distinguishable - so use these summer and fall escapes to help make decisions for the following season.

Summary

Monitoring growth stage is very important in order to obtain adequate control. Monitoring hairy fleabane development will minimize herbicide use while maximizing control. During warm winters, start monitoring in December. During most winters, monitoring should start in mid February to identify growth stage in order to properly time applications. Typically, preemergent applications made prior to mid February would not require a postemergent herbicide in order to control hairy fleabane. Preemergent applications made during February should be accompanied by monitoring in order to determine if hairy fleabane has emerged. If hairy fleabane has emerged, a postemergent herbicide must be added. Later winter and spring postemergent applications should be timed to early stages of development in order to minimize the amount of herbicide required and to ensure adequate control.

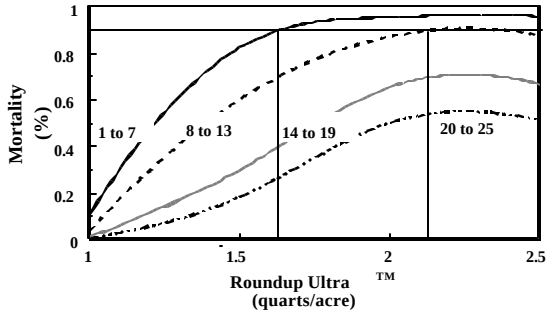


Figure 1. Amount of glyphosate (Roundup Ultra) required to control hairy fleabane across a range of growth stages represented as number of leaves per plant. Growth stages were 1 to 7 leaves, 8 to 13 leaves, 14 to 19 leaves and 20 to 25 leaves.

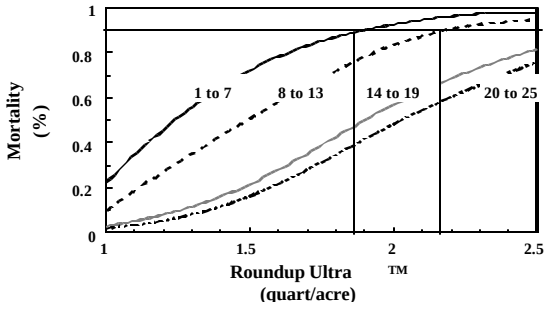


Figure 2. Amount of glyphosate (Roundup Ultra) required to control horseweed across a range of growth stages represented as number of leaves per plant. Growth stages were 1 to 7 leaves, 8 to 13 leaves, 14 to 19 leaves and 20 to 25 leaves.

IMPLEMENTATION AND VALIDATION OF A THERMAL DEATH DATABASE TO PREDICT EFFICACY OF SOIL SOLARIZATION FOR WEED MANAGEMENT IN CALIFORNIA

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Introduction

With the impending suspension of methyl bromide scheduled for 2005, the urgency for implementation of useful, alternative methods of soil disinfestation has never been greater. One of the fears is that when methyl bromide is no longer available, growers will resort to treating soil with "cocktails" of less effective, yet more environmentally hazardous chemicals. Implementation of effective, non-chemical soil treatments for use in high-value horticultural crops is critical. Soil solarization is a nonchemical, hydrothermal process which was developed to a large extent in the inland valleys of California. Solarization is now being used commercially in California on a limited, but increasing scale. Within its climatic limitations, it is an effective, broad-spectrum soil disinfestant for shallow-rooted crops and nursery production.

Although solarization has been shown to be an effective, broad-spectrum disinfestant in diverse agricultural systems, implementation has occurred at a relatively slow pace. There are several reasons for this, including ready availability of established fumigation methods, expert applicators, and technical support; as well as lack of treatment guidelines for users of solarization. Currently, treatment guidelines for solarization are limited to "treat for 3-6 weeks during the hot summer months". At a treatment cost of $250-400 per acre, and without the technical support they are used to receiving from chemical company reps, many growers have not felt confident enough to invest in solarization. This research will give users throughout California the ready availability of treatment guidelines for several important weed pests, which will be computer-linked to accumulation of heat dosage (temperature x time) in soil. Allowing users to have a good idea of when sufficient dosage units have been accumulated to terminate treatment on a pest-by-pest basis will greatly facilitate adoption of solarization.

This project was designed to provide an amplified database of thermal death dosages for weed pests of agricultural crops for California growers and pest control advisors wishing to use soil solarization.

Materials and Methods

Weed seed source: The six weed species used were *Echinochloa crus-galli* (barnyard grass), *Sisymbrium irio* (London rocket), *Portulaca oleracea* (common purslane), *Solanum nigrum* (black nightshade), *Sonchus oleraceus* (annual sowthistle), and *Amaranthus albus* (tumble pigweed). The species were selected to represent a range of thermal sensitivities and summer vs. winter growing habits. All seed were collected from plants growing wild in the Fresno area by Mr. Meso Beta.

Preparation of seeds for heating. Seeds from each species were placed in 3.5 cm diameter, nylon organdy bags. Each bag contained the number of seeds needed for ten to germinate, determined from the percent germination of the seed lots. Bags of seeds were dipped in deionized water and placed between moist paper towels in a crisper to imbibe water for 24 hours before the heat treatment. The only exception was common purslane, which was dipped 2 hours before treatment because it started to germinate within 24 hours. Bags were buried at a depth of 7 cm in sterile sand wetted to field capacity (10.4 % moisture) with deionized water in pint canning jars. The tops of the jars were covered with pieces of clear plastic tarp and sealed with the jar lids.

Treatments. Temperatures used were 39, 42, 46, 50, 60, and 70° C. Water baths were heated with a Fisher Isotemp immersion circulator (model 730). Six jars per species were heated at each temperature. Three unheated jars per species were kept in a water bath at room temperature (21° C) for each experiment as controls. Bags were extracted from the sand at 5 or more time intervals, which varied for each species depending on its susceptibility to heat. At each sampling time interval, one bag was taken from each jar. Temperature inside the bags was monitored with HOBO data loggers. Time intervals were initiated at the time that the temperature inside the bags reached the temperature being studied, which was consistently about a half- hour after the jars were placed in the water bath.

At 70° C, the method was changed because the sampling times were much shorter than the time it would take the jars to reach temperature. The jars were filled with the wet sand, closed, and placed in the water baths before adding the bags of seeds. The bags were buried in the jars when the sand reached 70° C and allowed to equilibrate for 6 minutes before starting the clock.

Germination tests. After being withdrawn from the jars, seeds were taken out of the bags and placed in 100 x 15

mm petri dishes on 7 mm diameter Whatman 1 filter paper moistened with 1.4 ml deionized water. The petri dishes were placed in crispers and incubated on a cycle of 8 hours at 20° C and 16 hours at 30° C, and exposed to a fluorescent grow light for the 16 hours at 30° C. Percentages of germination were determined after 14 days of incubation. Water was added to the petri dishes as needed during the 14 days. Seeds were counted as germinated if the radicle had emerged and the plumule had emerged to a length of 3 mm. A tetrazolium test was done on seeds that did not germinate to verify that they were dead (not feasible on common purslane and tumble pigweed). The percent germination from each bag of seeds was divided by the average percent germination of the three controls to correct for any variables besides temperature.

Results and Discussion

The six species can be separated into two groups by their ability to survive at high temperatures. Barnyardgrass, London rocket, and annual sowthistle died much sooner at each temperature than did black nightshade, common purslane, and tumble pigweed. In general, tumble pigweed was the hardest species to kill, and annual sowthistle was the easiest to kill.

At 70° C, seeds of all species were dead within 20 minutes. Barnyardgrass, London rocket, and annual sowthistle were dead within 10 minutes.

At 60° C, seeds of all species were dead within three hours. Barnyardgrass, London rocket, and annual sowthistle were dead within fifteen minutes. Tumble pigweed was dead within one hour, black nightshade was dead within two hours, and common purslane was dead within three hours.

At 50° C, thermal death occurred at a range of 4 hours (annual sowthistle) to 113 hours (tumble pigweed). London rocket was dead at 6 hours, barnyardgrass was dead at 9 hours, and black nightshade was dead at 71 hours. Common purslane was down to an LD_{90} by 23 hours, but germination was still not down to zero by 56 hours.

At 46° C, thermal death occurred at a range of 15 hours (annual sowthistle) to 13 days (tumble pigweed). Barnyardgrass was dead at 16 hours, London rocket at 24 hours, and black nightshade at 213 hours. Common purslane could not be studied at this temperature, because it began to germinate inside the bags during the heat treatment.

At 42° C, barnyard grass, tumble pigweed, and common purslane germinated inside the jars during the heat treatment. Black nightshade died within 16 days.

At 39° C, barnyard grass, tumble pigweed, and common purslane were not studied, since a temperature of 42° C had no effect on their germination. London rocket seeds were infected with a non-surface sterilizable fungus that made it impossible to do a thermal death study. Black nightshade germination was still high after 28 days of heat treatment. In contrast to earlier temperatures, the black nightshade seeds began to germinate soon after being removed from the heat.

Conclusions

Thermal death time requirements for the six weed species studied varied by as much as two weeks at given temperatures. These differences in heat dosage requirement among weed species could result in inadequate duration of solarization treatment for the more thermotolerant species. Time of year and variability of weather conditions over space and time during solarization treatment also could confound accurate determination of correct heat dosage. To overcome these variables, thermal death equations for specific weed species will allow users of solarization to know how long to treat their soil.

PERSPECTIVE: "MEANS WERE NOT SIGNIFICANTLY DIFFERENT, BUT . . ."

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As a frequent reviewer of manuscripts, I have noticed an alarming trend that seems to be growing. There is a decided tendency on the part of many authors to use a phrase, or a slight variation thereof, that goes something like this: "Means were not significantly different (P = 0.05), however plots treated with 'disodium bubbly muctate' produced numerically higher yields than did the untreated control." Equally troubling is the statement: "Differences were not significant, but there was a trend for . . ." I hear similar statements being made at meetings, both professional and clientele oriented. Such statements are scientifically incorrect at best, and misleading at worst. In the case of the first example, such a statement conveys the troubling message that disodium bubbly muctate really did increase yields when in fact, the differences observed are due to chance alone and not the action of disodium bubbly muctate. In the second example, one is again led to believe that the treatment actually produced some real

result when, again, the differences observed were due to chance. This can be very misleading to growers, PCAs and others who may not totally grasp the meaning of statistical significance.

Statistics deals with the relation between populations and their samples and the ultimate objective of statistics is to draw inductive inferences about the populations from given samples (Li, 1964). The level of significance is really the probability of committing a type I error. A type I error is defined as the rejection of a hypothesis that is actually true and conversely, a type II error is defined as accepting as true a hypothesis that is actually false (Li, 1964). Theoretically the significance level is arbitrarily chosen, but in practice 5% and 1% significance levels (or α) are usually used. A few years ago, the only tables available were for the 5% and 1% levels of probability. Current computer statistical packages usually calculate exact probabilities, however we still generally use the 5% or 1% levels. Our hypothesis (H_O) is generally that there is no significant difference between the means i.e. $\mu 1 = \mu 2 = \ldots = \mu k$. If the "F" value from our analysis of variance (ANOVA) exceeds the tabular value, or if the computed probability is <0.05 (or 0.01), the F value is significant at that level of probability; i.e. we reject H_O. By selecting the 5% level of probability, we agree that we are willing to accept a 1 in 20 chance of making the wrong decision regarding our hypothesis i.e. committing a type I error. If F is not significant, then the difference among the treatment means may be due to chance alone and not to any treatment(s) applied. Having made this decision, it should be adhered to, and no attempt to make more out of the data than exists should be made.

Dr. Werner J. Lipton, ASHS Science Editor, made this point extremely well. He wrote, "If you cannot tell whether three mountain peaks differ in height, can you legitimately call one the highest?. . . Similarly, if you have three data points that are not identical in value, but that do not differ significantly according to your statistical analysis, can you legitimately claim that, for example, treatment B produced the longest bean pods? I do not think so. After all, according to the probability you chose, the difference may be due to chance alone. *Once you set your limits, you cannot ignore them.*" (Lipton, 1991).

Many researchers are so convinced that a treatment or treatments are effective that even in the face of statistical evidence to the contrary, they cannot admit that the treatment had no effect and thus some parenthetical statement regarding the treatment effect creeps into the presentation of results. This does no one any favors, least of all our clientele who we are trying to serve. The next time you are tempted to discount the results of a statistical analysis ask yourself the following question . .. "Would I want the medical professional who tested the lifesaving medication I am now taking to have made this same decision because he felt so strongly about the 'drug'?" Let's all be more careful regarding such statements.

References Cited

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