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Inside

Sanitation to Reduce Arthropod Pests

by James Bethke

As you can imagine, my colleagues and I have witnessed all manner of cleanliness in ornamental plant production throughout the years. When beginning a new crop cycle or moving production into new facilities, it's easy to overlook the importance of cleanliness and just start filling benches. And if you been successful in growing a crop like poinsettias in the same house for years without considering sanitation, you may not think it's necessary to change your practices. However, not implementing a sanitation program is the most common mistake made by growers and can have dire consequences down the road, especially for those who plan to produce herbs or organic food products. It should be obvious that if you can significantly reduce or exclude pests and damage by starting "clean" (pest and disease free) and maintaining a high level of cleanliness, you will reduce the need for pesticides and have a higher quality of product. Although there are other factors that determine quality and crop yield, the level of sanitation will have great impacts.

Editor's Note

To survive in an increasingly competitive marketplace, greenhouse and nursery businesses must find ways to cut costs without undermining crop quality. One of the best ways to reduce pest management costs, while actually increasing crop quality, is to implement a sanitation program. Sanitation is the focus of this newsletter issue and is the first part of a multiple-part series on IPM. We begin this series with sanitation because starting clean and keeping clean is a key component of IPM and is a critical first step in managing pests. In addition to our regular columns, included in this issue are three feature articles by Bethke, Tjosvold and Wilen that focus on general sanitation practices for managing pests and diseases. Treatments to sanitize used pots (Suskow and Kosta) and recycled water (Oki, Merhaut) for disease prevention are also discussed. And in "Science to the Grower," Evans humorously finds a link between the sanitation theme and the way *Phytophthora* zoospores communicate.

♦ Steve Tjosvold and Julie Newman

Sanitation to Reduce Arthropod Pests

continued from page 1

When I was in college, I had a job sanitizing cleanrooms in a heart valve production company. There were various levels of sanitation and cleaning that were required in this operation, and each level of sanitation was monitored on a daily basis using sophisticated equipment. The highest level of clean was required in the area where the heart valves were assembled. Every day, every square inch of space in the room had to wiped with concentrated disinfectant and it was also necessary to spray the room with an aerosol disinfectant each time someone left the room. Filtered air flowed into the cleanrooms and out into the area outside the cleanrooms, which required the second level of clean. Obviously, the office spaces did not require as high a level of clean as the lab spaces and areas adjacent to the labs. Clearly, sanitation was a high priority for this operation. The heart valves they produced were of the highest quality and were in great demand.

Complacency is the elephant in the room. Complacency is when someone is self-satisfied while at the same time unaware of or ignoring actual threats, and in this industry, it puts plants at risk. Sanitation is the foundation of all pest and disease management programs in commercial floriculture and nursery crops, and you will hear this word in most presentations at horticulture conferences when pest and disease control is discussed. Just as in the heart valve production facility, the level of effort you put into being clean will have an effect on the level of pest management you will eventually have to employ.

Fig. 1. The greenhouse workers at this facility have to follow a high level of sanitation and strict entry requirements, including double entry doors (anteway), covering clothes that may harbor thrips and other pests, and stepping into a footbath to sanitize footwear. *Photo*: J. Bethke.

Even in ornamental plant production, there are obvious levels of clean required (e.g., high level of sanitation and clean, fig. 1). Not every facility will have the same sanitation issues or utilize the same best management practices to address cleanliness. In addition, there is a great variety of production types in ornamental plant production and type of production also affects the level of clean required. If a crop is going to be on site for a lengthy period of time, the chances of exposure to pests and pest proliferation is high and will require a higher level of sanitation. Regulatory issues also affect the level of clean required. If you are shipping out of the county, state, or country, you will most likely need a much higher level of clean due to phytosanitary requirements; if you are growing a crop that is a host plant for a regulated invasive pest, quarantine measures necessitate that the level of clean be at the highest level.

Exclusion

Excluding pests is an effective method of maintaining a pest-free greenhouse or production facility, and exclusion screening should be considered, especially if the commodity you are producing is susceptible. For example, years ago it was very difficult to grow impatiens without excluding thrips because they vectored tomato spotted wilt virus (TSWV) and impatiens necrotic spot virus (INSV). Growers can successfully exclude many arthropod pests by screening the pads at the end of the greenhouse where air is drawn in through the fan-and-pad cooling system (fig. 2). Additionally, pests can be excluded from greenhouses by creating a double door entry (anteway) where one door does not oppose the other. Inside the anteway, sticky cards or air curtains can also be used to assist exclusion of arthropod pests.

When Plants or Propagated Cuttings Arrive

One of the most important things you can do is to make sure that you are starting with clean plants or propagated material. Inspecting propagated material and maintaining pest-free stock plants for propagation will ensure a clean start and prevent pest populations from spreading (fig. 3). Additionally, if

Sanitation to Reduce Arthropod Pests

continued from page 2

Fig. 2. One end of this greenhouse is screened to exclude pest entrance through the pad end of a fan-and-pad cooling system. *Photo:* J. Bethke.

you bring in larger plants for finishing, isolate the introductions until you are sure they are pest free before you introduce them into a clean production area alongside other plants.

The cleaner you start a production area, the cleaner it will most likely remain. It is very important to remove all old crop residues and weeds, which may be harboring insect pests. If necessary, sterilize the growing environment with a disinfectant prior to new plant introduction to reduce or eliminate existing or senescing pests, those waiting for the next crop.

Fig. 3. Azalea cutting with chlorotic spots indicating a mealybug infestation. Inspecting mother stock plants and the production area of stock plants that harbored this infestation before taking cuttings could have prevented the spread of this pest. *Photo*: J. Bethke.

As an example, we have experienced several cases of excessive numbers of fungus gnats and millipedes attacking crops on raised benches. These pests were reproducing in great numbers in the peat that had been building up under the benches for a long time. Another example is a greenhouse where seeds and propagated material were growing under the benches and harboring mealybugs and the European pepper moth. Needless to say, these problems could have been avoided with good sanitation practices.

Hitchhikers

Arthropod pests can ride on containers (fig. 4), soil, equipment, new plants and people. Therefore, anything that enters a production area should be cleaned, sterilized or isolated before introducing them into a cleaned environment. Soil and other potting media should be sterilized, and soil that is not used right away should be kept tarped or in enclosed containers to keep it clean. Carts, tractors, trucks, etc. should also be cleaned before entering a clean production area. Workers can be a source of arthropod pests. Are they wearing brightly colored clothing? Have they just come from a pest-infested area? Have your workers just moved older plants with pests and are now entering a house with newly potted plants?

Monitoring

After a clean start to ensure cleanliness you will need to do a good job of pest monitoring. Early detection of a problem leads to an easier solution and it maintains a clean growing environment to produce a high quality plant. As I mentioned above, the heart valve company monitored each level of clean with sophisticated equipment, but it doesn't take a lot of sophistication to monitor for pests and diseases.

Training, Assessments and Record Keeping

In a good sanitation program, commitment is key and employee behavior is worth examining. Have your workers become complacent? Conduct routine self-assessments to eliminate complacency. It pays to periodically scrutinize aspects

Sanitation to Reduce Anthropods

continued from page 3

of your production and production facility because good sanitation requires a conscious effort on the part of every employee. Managers should implement programs to keep sanitation on the forefront of their job and they should remind workers about the importance of sanitation. This can be accomplished by sending out memos, hanging posters, sending texts or emails to employees or holding weekly team meetings. In addition, have you considered a sanitation training program tailored to fit your working environment? I am sure your workers are required to take scheduled training classes. Consider adding a short module about the importance of sanitation. Periodically reassess progress in sanitation practices at your facility. Keep records of areas that need attention and follow up.

Fig. 4. Pots and flats should be washed and sterilized prior to use, especially if they are being reused, and plants should be examined for pests when received and prior to potting. *Photo*: J. Bethke.

Final Comments

Keeping something clean seems somewhat simplistic, but doesn't it make sense? I know from experience that a higher quality product can be produced from a clean and well-maintained growing environment. I also know that every grower, no matter how clean they are, can always improve. Hopefully, the information herein will help you take a fresh look at your property and take you to another level of *clean.*

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Keeping Weeds Out of Your Operation

by Cheryl Wilen

Weeds can easily infest a nursery or greenhouse if good sanitation procedures are not followed. In general, most problematic weeds are annuals and are introduced as seeds although some perennial weeds such as yellow nutsedge can be introduced as tubers if mineral soil or sand is used as a potting mix component.

Common nursery weeds are those that have seeds that are easily picked up by the wind such as common groundsel and northern willowherb and those that have a special mechanism that promotes the spread of the seeds. These include woodsorrel and bittercress species.

Keeping Weeds Out of Your Operation

continued from page 4

It is important to prevent these easily spread weeds from entering the growing area and to quickly remove them well before flowering. For wind-carried seeds, prevention starts by cleaning up the areas in and around the beds. Gravel beds should be floated between crops and nursery cloth should be inspected for holes and spilled soil. Removing the soil is a particularly important sanitation task as weed seeds can easily become established there and root into the nursery cloth, creating more holes when removed (fig. 1). Herbicides can be used to prevent weeds but care must be taken to choose ones that have high Koc values (herbicides that are strongly adsorbed to soil particles) or low solubility to reduce runoff or leaching.

Fig. 1. The potting soil on this nursery cloth should be removed because it facilitates the establishment of weeds that can root into the fabric. *Photo*: C. Wilen.

Within nursery containers, herbicides or mulches can also suppress weeds seed establishment. We have found that mulches are most effective in 5-gallon and larger containers, and a 1-inch layer of coarse mulch (with a particle size about 1/4 to 1/2 inch) is generally needed to get good weed control. When herbicides are used, the first application should be made as soon as possible after setting out the plants. The longer the plants are in the open unprotected from weeds blowing in, the harder it is to control weeds through the whole plant growing cycle.

For weeds that spread shorter distances by "popping" from pods such as woodsorrel and bittercress, the same precautions should be taken. However, these types of weeds often have rough seed coats that stick to plastic materials, including pots and irrigation tubing, and other surfaces (fig. 2). Where these weeds are found, part of the sanitation routine should be to use new pots or thoroughly clean used ones and wipe down benches and tubing. Of course, all areas should be monitored and weeds removed before they flower as the first step of prevention. Start clean and stay clean.

In greenhouses, weeds around the doors (fig. 3) should be

Fig. 2. Oxalis seeds can stick on plastic such as this watering can *(left)* and on other surfaces such as this greenhouse pole (*right*) serving as a source of inoculum for nursery plants. *Photo*: C. Wilen.

controlled, not only because seeds can get into the greenhouse and infest the pots, but also because weeds can harbor pest insects or plant pathogens that can easily move into the greenhouse. For similar reasons, weeds under benches (fig. 4) or growing along the inside walls should be cleaned up. Often the reason weeds proliferate in these areas is due to overwatering, resulting in overly wet and often highly fertilized places that promote weed growth. Modifying irrigation method and amount and improving drainage will greatly reduce weed growth.

Pulled weeds should be removed from the growing area and placed in covered trash containers. Some weeds will root

Keeping Weeds Out of Your Operation

continued from page 5

from stems left on the ground and flowers may continue to mature on some plants (see video at http://ucanr.edu/blogs/blogcore/postdetail.cfm?postnum=17079) which may produce viable seeds.

Fig. 3. The weeds growing outside this greenhouse door should be removed because they can spread weeds and other pests inside the greenhouse where they can infest nursery crops. *Photo*: C. Wilen.

Other areas where sanitation is key include piles of potting mix or their individual components. Piles being stored for custom mixing tend to be great sites for weed seeds to land on, grow on, or even just remain dormant. This problem can be reduced by controlling nearby weeds and by using a tarp or plastic to protect the piles. Locating the storage and mixing area away from areas where weeds are growing will also help. Try to use bulk potting mix or components quickly; the longer a pile sits the more chance there is that it will collect weed seeds.

Finally, observe liners for a few days after receiving or transplanting. If possible, keep them in a holding area to make sure that new weeds are not introduced from these liners. While most liners come clean, there are occasions where a change in environment will promote the growth of some weeds that were not obvious in the liner production stage.

Fig. 4. Weeds growing under greenhouse benches and along walls can infest nearby plants. *Photo:* C. Wilen.

Adjusting irrigation to reduce runoff, using mulches and appropriate timing of herbicide applications will greatly reduce weed pressure. Concurrent use of sanitation methods, such as being diligent in weed removal in and around beds and greenhouses, under benches and near potting media storage, will contribute to a more sustainable weed management system.

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Sanitation for Pathogens: Contaminated Soil is a Primary Source of Pathogens and Must be Managed

by Steve Tjosvold

Maintaining good sanitation is one of the most important ways of preventing and managing diseases. Sanitation should be practiced during all phases of production. Practice good sanitation with planting materials, containers, media, irrigation water, benches and growing areas, tools and other equipment, and how workers use and work with these items. Because soil and associated plant debris can be a main source of pathogens, this article will focus on ways to manage contamination from these sources.

Pathogens can be found on the ground and non-sanitized container media, crop debris in soil or potting media, and anything that has contacted the ground, such as equipment, tools, irrigation hoses, and workers' hands and shoes. Containers should not sit directly on the ground. Many root infecting pathogens can be moved with water from a contaminated pot or area on the surface of the ground and infect roots of a nearby plant (fig. 1). Likewise during a rain storm or, in some cases, during sprinkler irrigation, pathogens can be splashed from the contaminated ground or pot onto nearby plants. Benches or similar structures that support plants above the ground can eliminate or minimize this. In greenhouse structures, concrete floors or other impervious surfaces are ideal for walkways between benches. Floor surfaces should be kept clean of debris, soil, or planting media. After a crop cycle and between crops, benches should be washed clean of plant debris and soil. Benches should be allowed to dry because this can kill sensitive plant pathogens. Metal and plastic benches are ideal because they can be cleaned and sanitized readily (fig. 2). Wooden benches should be regularly painted with copper-containing paints to eliminate fungi or other types of pathogens (fig. 3). Benches can be sprayed with diluted chlorine bleach (0.5 % sodium hypochlorite solution) or other

Fig. 1. Phytophthora root rot infection can be avoided with good management practices addressing soil sanitation. *Photo*: S. Tjosvold.

Fig. 2. Metal benches and concrete flooring are ideal for greenhouse sanitation because they are so easily cleaned and disinfected. *Photo:* S. Tjosvold.

Fig. 3. Wooden benches can be disinfected with copper containing paints to disinfect the wood and also extend its life. *Photo*: S. Tjosvold.

Sanitation for Pathogens: Contaminated Soil is a Primary Source of Pathogens and Must be Managed

continued from page 7

suitable disinfectant. This should be done after the benches have been thoroughly cleaned of all potting media and plant debris that would inhibit the activity of most disinfectants. Benches should be allowed to dry before they are used again. There are many clever ways to raise pots and containers off the ground (fig. 4 and 5). Where benches or other support

Fig. 4 (*bottom left*). Proprietary container supports pots elevated above the ground to prevent water runoff from infecting roots. Here the containers are placed on benches. *Photo*: S. Tjosvold.

Fig. 5 (*above right*). Tree pots can be cleverly placed inside larger pots to raise their bottoms off the ground. *Photo:* S. Tjosvold.

are not feasible for cost or other reasons, the soil should be covered with gravel and weed cloth. Gravel supports containers above the drainage water; the weed cloth (which also reduces weed germination) helps reduce splash and can be cleaned of soil and plant debris between crops (fig. 6).

Pots, flats, tools (fig. 7) and irrigation equipment such as piping or emitters can have clinging contaminated soil and plant debris and should never be reused without thoroughly washing them to remove all clinging particles. They can then be treated with a disinfectant such as diluted chlorine bleach. The bottom of clean shoes can be sanitized with disinfectants such as quaternary ammonium compounds (fig. 8).

Fig. 6. Weed cloth over gravel is a cost effective way of reducing pot contact with the ground and water runoff. *Photo*: S. Tjosvold.

Fig. 7 (*below left*). Tools must be free of clinging soil and debris before they are disinfected with chlorine bleach. *Photo:* S. Tjosvold.

Fig. 8 (*top right*). The bottom of shoes can sanitized if they are free of soil. *Photo*: S. Tjosvold.

Fig. 9 (*above*). Portable steam generator for field or large area use. *Photo*: S. Tjosvold.

Potting media should not be reused, as the risk of pathogen carryover is too great and not worth the savings in most cases. Even some newly formulated mixes may benefit from

Sanitation for Pathogens: Contaminated Soil is a Primary Source of Pathogens and Must be Managed

continued from page 8

steam treatment if the sources of the media are unknown or unreliable. Contact the soil formulator of these products to insure they are free of pathogens. Aerated steam is the most efficient way of sanitizing potting media. Portable steam generators are available for sale or rent for small batches, and commercial greenhouses may use the steam from boilers used for heating greenhouses (fig. 9 and 10). For most

Fig. 10. Steaming raised beds with the boiler used for heating the greenhouse. *Photo*: S. Tjosvold.

situations, aerated steam treatment of 140° to 160°F for 30 minutes will kill the most problematic pathogens while preserving the natural microflora of the growing medium. Store and handle potting media so it does not come in contact with the ground or water runoff. Cover the media when not in use (fig. 11).

Fig. 11. Cover or seal potting mix and other growing media when not in use. Make sure potting mix does not contact the soil and is protected from water runoff during rain events. *Photo*: S. Tjosvold.

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Solarization: A Simple and Low Cost Method for Disinfesting Horticultural Containers

by Karen Suslow and Kathy Kosta

The reuse of dirty plant pots by nursery growers has repeatedly been shown to be a method by which plant pathogens are transferred within or to a nursery. More critically, this practice is an efficient pathway to infest landscape setting or habitat restoration sites by the out-planting of pre-symptomatic infected plant material. The transfer of water molds (Oomycetes), such as plant pathogenic *Phytophthora* species, is a major threat to nursery operations and prevention of cross-contamination via pots should be a critical nursery management goal. Our research has established performance and efficacy criteria that demonstrate the risk can easily be managed by the solarization of used pots.

Solarization: A Simple and Low Cost Method for Disinfesting Horticultural Containers

continued from Page 9

Material and Methods

In the summer of 2015, the National Ornamental Research Site at Dominican University of California (NORS-DUC) conducted outdoor solarization experiments designed to determine the temperature and time requirements at which *Phytophthora cactorum*, a commonly found soilborne plant pathogen in the nursery industry, would be killed. *P. cactorum* served in our trials as a surrogate, a test organism to determine the efficacy of sanitation regimes for *P. ramorum* (which causes sudden oak death) and *P. tentaculata*, plant pathogens which were shown to be killed at similar disinfection temperatures as *P. cactorum*.

We used 1-gallon (1G), D-40 and Tubex Tubes, containers that are commonly used by small-scale and large-scale growers. Tubex Tubes are used to protect outplanted stock in restoration sites from browsing wildlife. In this trial, "treatments" are those containers wrapped tightly in clear polymer (plastic), bought off the shelf at a mass merchant store (fig. 1). "Controls" are the containers not wrapped in plastic. Trials were set up in two outdoor locations in California: in a hot climate (Winters, fig. 2) and in a cool climate on the coast (Pacifica).

Fig. 1. "Clear" polymer-wrapped 1G treatment. *Photo:* K. Suslow.

1G pots were nested 3 feet high and three stacks were secured together flat on the ground with gardening tape and laid on black plastic. Spectrum WatchDog Data loggers were inserted in the center stack, on the bottom-side of the centrally located pot, where we have demonstrated the coolest temperatures occur in the stack. When replicating this experiment at your facility, always ensure the data logger or temperature probe is in the coolest location so that the entire stack reaches the required temperatures. All pots in the treatments were sprayed with water prior to sealing them in plastic.

Fig. 2 (*left*). Field layout of treated (pots wrapped in plastic) and controls (pots not wrapped in clear plastic) at the Winters site. *Photo*: K Suslow.

Leaf disks from *P. cactorum*-infected rhododendron leaves (fig. 3) were mixed with soil and placed inside narrow mesh sachets; three sachets were then inserted into a hollow-core rope (fig. 4), spaced 8 inches apart for easy weekly retrieval. The experiment was conducted over a three-week period. Each week, one sachet was retrieved by extracting the rope through a hole drilled in the side of the nested 1G pots (fig. 5). The data loggers were located next to the sachets. The leaf disks were plated out to determine if the

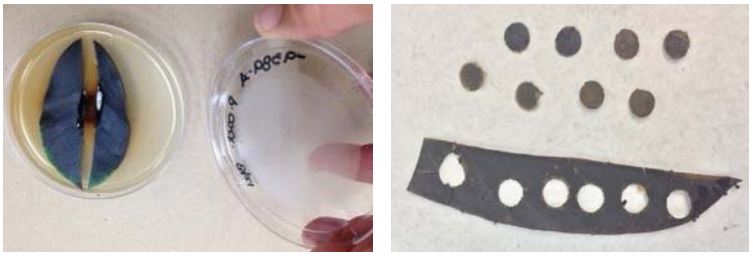

Fig. 3. *P. cactorum*-infected rhododendron leaves (*left)* were used to make leaf disks (*right*) that were mixed with soil and placed inside mesh sachets. *Photo:* S. Sharma.

Fig. 4 (*above*). The infected leaf disks and soil in sachets were inserted into hollow core rope. *Photo:* K. Kosta.

Fig. 5 (*above*). A hole drilled in the side of the nested 1G pots enabled weekly extraction of sachet. *Photo:* K. Suslow.

Solarization: A Simple and Low Cost Method for Disinfesting Horticultural Containers

continued from page 10

pathogen survived. In addition, lab-maintained *P. cactorum*-infected leaf disks in sachets were plated out weekly to ensure continued viability of the pathogen during the course of this experiment.

Results and Conclusions

For all pot sizes and in all trials, pots laid horizontal onto black plastic on the ground yielded higher daytime and nighttime temperatures than the ambient temperatures. Although reaching the required temperatures for solarizing is more easily attained in the summer months, in cooler climates and in the later fall months, the radiant heat from the ground and the black plastic will aid in hastening the solarizing process. The treatments (those pots wrapped in clear plastic) can obtain a heat-capture gain of up to 200°F (120°C) as compared to the controls (pots not enclosed in a plastic).

Table 1. Cumulative hours attained during the first week of the Winters and Pacifica trials for 1G pots. Data after the first week is not shown because the pathogen was killed in the treated pots during the first week in both research sites.

Winters, CA **Ambient temperature:** **(86–106^{0}F (30–41^{0}C)**	**104–113^{0}F (40–45^{0}C)**	**115–122^{0}F (46–50^{0}C)**	**≥ 124^{0}F (51^{0}C)**	**Pathogen killed**
Treatment	12 hrs	14 hrs	25 hrs	YES
Control	30 hrs	2 hrs	0	YES
Pacifica, CA **Ambient temperature: 66–79^{0}F (19–26^{0}C)**				
Treatment	18 hrs	0	0	YES
Control	0	0	0	NO

In lab studies, *P. cactorum*, as well as numerous other *Phytophthora* and *Pythium* species, can be killed at 120°F (50°C) for 30 minutes when exposed to moist heat (Baker, KF and Cook RJ; Griesbach et al.). Reducing the temperature and extending the time has proven to be just as effective at killing *P. ramorum*. In infected rhododendron tissue, in loam soil, *P. ramorum* was not recovered after two days at 104°F (40°C) nor at 4 days at 95°F (35°C) (Tooley et al). As noted in table 1, *P. cactorum* was killed in the treatments in the cool climate trial in Pacifica when the temperatures reached 104–130°F (40–45° C) for 18 hours cumulatively over the first week of the experiment. Ambient temperatures did not exceed 79°F (26°C).

When solarizing used 1G pots in the warm months of the year, wet the stacked pots, seal them in plastic and align them in a single layer on the ground on a black tarp so that they are exposed fully to the sun. With a temperature probe in the coolest part of the stacked pots, confirm temperatures have reached 122°F (50°C) for minimally 30 minutes; this can potentially be achieved within a day in a warm climate. Without a temperature probe, track the ambient temperature and when it reaches 106°F (41°C) on a daily basis for a week, from the data in table 1 you can expect that your sealed pots have attained the necessary temperature to kill *Phytophthora* and *Pythium* species.

Future pot solarization work at NORS-DUC includes using the quarantine pathogens *P. ramorum* and *P. tentaculata*, trialing pot solarization on a large scale with a commercial nursery and configuring the pots in different layouts to increase the throughput.

Solarization: A Simple and Low Cost Method for Disinfesting Horticultural Containers

continued from Page 11

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Slow Sand Filters Remove Tobacco Mosaic Virus *by* Loren Oki, Lloyd Nackley and Bruno Pitton

Slow sand filters (SSF) are an old technology that utilizes a microbial community to degrade contaminants in water, including plant pathogens. The uncleaned water passes through a bed of sand that serves as a substrate for the microbial community, allowing a biofilm to form around the sand grains. As the filter's microbial community matures, the surface of the sand bed becomes covered with a thick layer of microbes commonly referred to as the "schmutzdecke," German for "dirt layer." The exact treatment mechanisms are unclear, but plant pathogen removal is linked to the schmutzdecke. However, some removal of larger plant pathogens and other contaminants may be due to either physical filtering or adhesion to the sand itself. The properties of the sand bed and filtration system are important the to optimize filtration process:

- The sand should be uniform round grains about 0.3 to 0.6 mm in diameter.
- The sand bed should be approximately 3 feet deep to allow for removal of the top few centimeters of sand once the biofilm has thickened and flow is restricted.
- There should be about 3 feet of water above the sand bed to provide pressure, as gravity is the means for water flow.
- Controlling the flow rate through the sand bed is important to optimize treatment. If the flow rate is too high, treatment is ineffective.
- Pre-filtration of particulates prior to treatment will help prevent sand bed clogging and extend intervals between maintenance.

Slow Sand Filters Remove Tobacco Mosaic Virus

Continued from Page 12

As the name implies, flow rates through the sand bed are slow. One square foot of sand bed area can treat 0.06 to 0.2 gallons per minute. So, a 12-foot diameter round tank containing a sand bed would be able to treat about 10,000 gallons per day at the lower flow rate. The containment used to hold the sand bed is not important and could be a water tank, septic tank, or lined reservoir, for example. A manifold is placed at the bottom of the containment to collect the treated water and is covered with pea gravel. Above the pea gravel is a series of layers of graduating sand size, from coarse to fine, to prevent the fine sand from entering the pea gravel. Above these layers is the filtration sand bed material. A pump is used to extract the treated water from the sand bed system into a storage tank.

Fig. 1. Former Graduate Student Mike Harris is adjusting flow rates on slow sand filters during an experiment studying the removal of *Phytophthora capsisi* from captured runoff water. *Photo*: L. Oki.

Our previous work (Lee and Oki 2013) showed that SSFs are effective at removing *Phytophthora* when captured water was inoculated with the pathogen (fig. 1). However, Deborah Mathews, a plant pathologist formerly at UC Riverside, wanted to know if these filters could remove plant pathogenic viruses. The literature was unclear if SSFs could remove plant viruses (Krczal et al. 1995; van Os et al. 1998), although extensive work shows removal of viruses that affect bacteria and humans (Paranychianakisa et al. 2006). A pilot study was conducted at the UC ANR South Coast Research and Extension Center in Irvine, CA using a single sand filter made of 4-inch PVC pipe to see if Tobacco mosaic virus (TMV) removal was effective. Anaheim pepper plants were irrigated on benches in a greenhouse and the runoff was captured and pumped to the top of the filter system.

Before inoculation of TMV, water samples from the filter were retrieved from above the sand bed (pretreatment) and after the water had passed through the bed (post treatment) to get background measurements (Day 0). After the water samples were collected, a suspension of TMV was added to the water at the top of the filter three days per week. Water samples were collected 24 hours after the initial TMV addition and at 7-day intervals thereafter for a total duration of 12 weeks. The water samples were analyzed using ELISA and bioassays in which *Nicotiana glutinosa*, *Chenopodium quinoa, N. benthamiana* and *N. tabacum* were inoculated with pre- and post-treatment water and evaluated for symptoms. *N. glutinous* and *C. quinoa* were local lesion/hypersensitive response bioassays, whereas *N. benthamiana* and *N. tabacum* were systemic whole plant assays.

Both the ELISA and bioassays showed that the TMV was removed after about 9 weeks. The pilot study was repeated using three filters, but only the whole plant assays were used along with the ELISA analyses. Again, TMV was removed from the filtered water after about 6 weeks (table 1.). The faster removal could be attributed to warmer temperatures as the second study occurred later in the season compared to the first.

Plant viruses, including TMV (Hong and Moorman 2005), have been recovered from irrigation runoff, so their spread through the reuse of this water is highly likely. TMV is known to be a very robust virus that can survive for very long periods in a wide variety of conditions and its removal means that the removal of other plant pathogenic viruses is likely.

Also see: Oki, L. 2013. Update of slow sand filtration research. UCNFA News. 17(2): 4-6. http://ucnfanews.ucanr.edu/Articles/Feature_Stories/Update_of_Slow_Sand_Filtration_Research/.

Slow Sand Filters Remove Tobacco Mosaic Virus

continued from Page 13

Table 1. Results of bioassays conducted on water samples collected from slow sand filters (SSF) post treatment, after the water passed through the filters. The Time 0 samples were collected before TMV was added to the SSFs. Plants used for the assays were *Nicotiana benthamiana* (N.b.) and *N. tabacum* (N.t.). At Week 6 and therafter, all of the assays were negative indicating no presence of TMV

	Column 2	Column 3	Column 4
TIME	N.b./N.t.	N.b./N.t.	N.b./N.t.
-0	-/-	-/-	-/-
24 hrs	+/+	+/+	+/+
Wk 1	+/+	+/+	+/+
Wk 2	+/+	+/+	+/+
Wk 3	+/+	+/+	+/+
Wk 4	+/+	+/+	+/+
Wk 5	-/+	+/+	+/+
Wk 6	-/-	-/-	-/-
Wk 7	-/-	-/-	-/-
Wk 8	-/-	-/-	-/-
Wk 9	-/-	-/-	-/-
Wk 10	-/-	-/-	-/-
Wk 11	-/-	-/-	-/-
Wk 12	-/-	-/-	-/-

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GET CULTURED: Sanitation and treatment for pathogens in recycled water

by Donald J. Merhaut

Water recycling conserves water and reduces irrigation and fertilizer costs. However, the presence of plant pathogens in recycled water such as *Phytophthora* and *Pythium*, as well as bacteria and viruses, can lead to the spread of diseases in the nursery with potential consequences. Cultural practices to prevent diseases in the nursery such as good sanitation practices are the most effective ways to reduce pathogens in recycled water. In addition, recycled water should be treated to kill pathogens as a standard sanitation and preventative control practice. There are several physical and chemical methods to treat recycled water. One method, discussed by Loren Oki in his feature article, is slow sand filtration. I will discuss the other methods for treating recycled water: chlorination, ultraviolet light, ozonation, copper ionization and heat treatment.

Chlorination

Chlorine kills organisms through the oxidation of cell membranes. The three methods of incorporating chlorine into irrigation water include sodium hypochlorite, calcium hypochlorite and chlorine gas. Always check with local agencies regarding the regulations associated with the use and storage of chlorine products.

Effectiveness of treatment depends on several factors, including water cleanliness, concentration of chlorine, pH and temperature. The dirtier the water, the more chlorine it requires to treat it, because chlorine binds to other mineral and organic particles. As the concentration of chlorine increases, the effectiveness increases along with the rate of disinfection. Most crops will tolerate 100 ppm chlorine. Certain pathogens require higher chlorine concentrations or longer exposure times for effective treatment.

When using chlorine, you should strive for a residual of 0.5 ppm available chlorine for water not containing ammonia. Water containing ammonium-N will form chloramines, and your total residual combined chlorine should range from 0.5 to 1.0 ppm. Typically the dosage ranges from 4.5 to 8.0 ppm. The residual will always be less than the dosage since much of the chlorine reacts with contaminants in the water. Too much residual chlorine can damage sensitive crops.

Chlorine molecules are most stable, and therefore most effective, at a neutral pH (7.0). Low water temperature (less than 50°F) or high temperature (greater than 68°F) may reduce chlorine effectiveness. The disadvantages of chlorine are that residual chlorine can damage or kill plants, and chlorine does not break down or remove most pesticides or herbicides. Also, brown coloration, due to dissolved organic matter and acids, will not be removed with chlorination. Moreover, the fumes are highly dangerous, leading to a worker/operator exposure concern.

Ultraviolet light (UV)

Ultraviolet light, which has a wavelength from 100 to 400 nanometers (nm), may be used to kill pathogens. The mode of action is the production of free radicals disrupt cell membranes and kill organisms. In order to be effective, the water being treated must be relatively clear and colorless. Nurseries that use hydroponic or soilless systems are candidates for UV treatments. Nurseries using traditional methods of producing plants in soil would need to subject the water to other filtration processes to remove any suspended fractions or discolorations in the water.

Four types of lamps can be used to emit UV: low-pressure mercury lamps, high-pressure mercury lamps, excimer lasers and xenon flash lamps. Low-pressure mercury lamps emit a wavelength of about 254 nanometers. High-pressure mercury lamps emit a wavelength of 190 nanometers, which also causes the formation of sanitizing ozone in the water. Excimer lasers emit pulses of light at 248 nanometers. Xenon lamps emit light over a larger spectrum, some of which is not UV; therefore, this light source is not as energy efficient.

There are several advantages of using UV for water disinfection. The cost of operation is low if the water source is relatively clean. No chemicals are used in this process, and very few components must be maintained. Finally, even though it kills all organisms in the water, there is no resulting residue that is toxic to plants.

A major disadvantage of using UV is that the water must be

GET CULTURED: Sanitation and treatment for pathogens in recycled water

continued from Page 15

fairly clear and clean of debris for effective treatment. If the water source is not clean, a UV-exposure time of longer than 30 seconds will be required for complete pathogen kill. Light sources may chemically denature some chelates that are used to keep micronutrients in the soluble form. UV does not remove other chemicals from the water, such as herbicides and pesticides. UV treatment also does not remove discoloration caused by organic acids.

Ozonation

Ozone is an oxidant that, like chlorine, kills organisms by disrupting cell membranes. The ability of any specific chemical to cause oxidation is measured as oxidation reduction potential (ORP). ORP values of 700 millivolts should provide complete disinfection. ORP values less than 300 millivolts are usually considered safe for most aquatic life.

The advantages of ozonation are that no residual chemicals remain after treatment, no chemical storage is required since ozone is manufactured on-site, the system efficiency is inexpensively monitored by measuring ORP values, and ozonation effectively oxidizes most pesticides. A major disadvantage of ozonation is that it requires a fairly clean water source to work properly. Therefore, water with large amounts of organic matter, clay, or other debris requires increasing the ozone exposure time (20 minutes to 1 hour). Other disadvantages of ozonation include an increased cost due to the use of electricity to produce ozone, the risk of some chelated nutrients precipitating out, and an increase in water pH, which may require acidification. Furthermore, ozonation is not completely effective in killing chlamydospores and microsclerotia of some pathogens.

Copper ionization

Copper ionization is the process of adding copper ions to water. Copper electrodes are inserted into the water, and an electric current is passed through the electrode, releasing copper ions into the water. Effective pathogen treatment requires an ion concentration of approximately 50 to 300 parts per billion (ppb). While copper ionization has been used to treat algae in greenhouse coolant pads, there has been limited use for treatment of irrigation water. In hydroponic systems, a copper concentration of approximately 50 ppb is recommended, which also contributes to plant nutrients since copper is a necessary plant nutrient. However, a related disadvantage is that certain crops are sensitive to the concentration of copper recommended for effective treatment, and toxic levels may accumulate in closed recirculating systems.

An advantage of copper ionization is the relatively low cost for installation and maintenance of the system. The copper ionization system is also portable, alleviating the need for large holding tanks; there is also no chemical storage necessary with this process. However, like many other disinfectants, the effectiveness of copper ionization is reduced if the water contains excessive amounts of organic matter, clay, or other debris.

Heat Disinfection

Heat treatment has also been successfully used by the nursery industry to sterilize soils and growing media. European countries use heat to sterilize water, and this method has also been used in California greenhouses (fig. 1).

Fig. 1. Before water is re-used on greenhouse crops in this nursery, it is heat-treated to reduce pathogens. These tanks store the heat-treated recycled water. *Photo:* J. Newman.

GET CULTURED: Sanitation and Treatment for Pathogens in Recycled Water

continued from Page 16

Viruses are killed at temperatures of 130°F, with an exposure time of 1.5 hours. As the temperature is increased, exposure time decreases. Heat exchangers (for heating water) are located at one point of the water system. Prior to heating, the water pH is reduced to approximately 4.5 to prevent calcium accumulation on the heat columns.

The primary advantage of heat treatment is that the water does not have to be as clean as is required in other chemical treatment processes. The other major benefit is that no chemicals are used, alleviating chemical storage and residue issues.

Some disadvantages of heat treatment include the following: water must be cooled before being applied to plants, heating water adds to the cost, heating duration may be as long as 1 hour, calcium removal from heat exchangers needs periodic maintenance, additional space is required for tanks to hold treated water, and high heat denatures chelates that are used for micronutrients.

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SCIENCE TO THE GROWER: Sync and Swim
Pythium and Phytophthora don't mind asking for directions

by Richard Evans

It's Summer! To celebrate, my wife and I took our grandkids to the public swimming pool, but the joy was quickly dampened (so to speak) by voices that started at one corner of the pool and traveled toward us in a wave. When the wave reached us, we learned that someone had discovered a turd floating in the water. We hopped out of the pool to escape the spreading cloud of microorganisms, and I took advantage of the down-time to ponder the aquatic pathogens that infest irrigation water and substrates in greenhouses an nurseries.

Most of the root diseases of plants growing in hydroponic systems or soilless substrates have been attributed to the genera *Pythium* and *Phytophthora,* which are fungus-like microorganisms that taxonomists have reclassified as oomycetes. In fact, they are more akin to aquatic organisms like brown algae than to fungi. Both *Pythium* and *Phytophthora* love aquatic environments, and they produce asexual single-celled zoospores that are outfitted with two appendages, called flagella, which they use to swim. One flagellum works like an oar to propel the zoospore through water. The other flagellum serves as a rudder for steering.

These zoospores serve as the primary dispersal and infection agents, and they can swim nearly 3 feet per hour. They may not be as fast as Mark Spitz, but they can beat anything Mark spits.

Researchers have known for many years that *Pythium* and *Phytophthora* have a "homing response" — they can locate targets for infection and swim toward them. Early evidence for this was the discovery that they follow chemical trails, a process called chemotaxis. Some of the attractive chemicals are nonspecific ones, like amino acids and ethanol that may be released by injured roots. Others are compounds unique to a particular kind of plant, and their presence, even at exceedingly low concentrations, enables the pathogen to identify and infect a host species. Some chemicals may even warn the pathogens of trouble ahead, causing them to turn and swim away, just as you might slip out the back door when you detect your mother-in-law's perfume in the foyer of your house.

Researchers have reported that plant infection by *Pythium* and *Phytophthora* usually doesn't occur unless a threshold

SCIENCE TO THE GROWER: Sync and Swim
Pythium and Phytophthora don't mind asking for directions

continued from page 17

density — on the order of millions of zoospores per ounce of water — is present. It appears they need to summon the troops in order to invade a plant, but it turns out that isn't necessarily true. Indeed, just like those humans who communicated the presence of something unpleasant in the pool, *Pythium* and *Phytophthora* can communicate with each other! Researchers at Virginia Tech (Kong and Hong 2010) found that *Phytophthora* zoospores (and perhaps other forms of *Phytophthora)* can communicate through a process called quorum sensing. The pathogen produces, and releases into the water, a chemical that can be detected by its neighbors, enabling them to count their numbers. Kong and Hong filtered a solution containing *Phytophthora* to isolate the zoospores from the chemicals they were releasing. They called this a zoospore-free fluid. Then they exposed plant tissue to a single zoospore. Infection rarely occurred in the absence of the zoospore-free fluid, but a single zoospore provided with the zoospore-free fluid almost always infected the plant.

Even more remarkable is that different species of *Phytophthora* and *Pythium* are able to communicate with each other. Hong's group at Virginia Tech found that zoospore-free fluids collected from several species of *Phytophthora* and one species of *Pythium* could stimulate plant infection by another *Phytophthora* species (Kong and others 2010). The researchers suggested that cooperative behavior among oomycetes gives them a competitive advantage over other groups of pathogens, especially when food is in short supply. This signaling among oomycetes also may explain why individual species of *Phytophthora* or *Pythium* sometimes cannot be detected before serious disease outbreaks occur.

Communication among oomycetes may also help to account for a strange phenomenon called pattern swimming. Despite their microscopic size, *Phytophthora* and *Pythium* zoospores can swim in synchrony, forming patterns that are visible to the naked eye. Speaking of things visible to the naked eye, I wonder if it's safe for me to hop back into the pool.

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DISEASE FOCUS: Powdery and downy mildews

by Jim Downer

Two kinds of "mildew" foliage diseases are commonplace in nurseries but are very different from each other biologically. While both kinds of disease cause damage and symptoms on the new growth, foliage and young stems of ornamental plants, the biology and thus control of these diseases are very different. Both pathogen groups are similar in that they parasitize living plants and generally are difficult to grow in culture plates, preferring a living host rather than artificial media for their growth. Powdery mildews are more common but we are seeing an insurgence of new downy mildews in the ornamental plants.

Powdery mildews are plant pathogens attacking over 10,000

DISEASE FOCUS: Powdery and downy mildews

continued from page 18

species of plants and belonging to more than 1600 genera of fungi in the class Ascomycetes. Many Ascomycetes including most powdery mildews are "compound interest" diseases meaning that they have two spore production phases: a sexual stage that usually supplies the primary (first infection inoculum) and a secondary conidial stage that produces asexual spores. The conidial stage is what we recognize as powdery mildew (fig. 1). Since all of the mildew spores look very similar mycologists called this part of the life cycle Oidium or the Oidial stage. Modern molecular biology techniques have helped identify and reclassify mildews that have never formed the sexual stage. The sexual part of the life cycle involves production of ascospores in fallen leaves inside a structure called a cleistothecium. Sexual spores are released in the spring during wet periods and are wind and splash borne onto new tissues where the first infections occur resulting in the mildew or conidial stages that can continue to reinfect all season long on new growth. Powdery mildew conidia only infect living tissues of epidermal cells and do not penetrate to deeper layers. They are obligate parasites or biotrophs, which require living host cells to grow in. Infection results in impaired growth, often resulting in cupped, curled or distorted foliage. These fungi can infect in moderately humid or even dry conditions. Free water is not necessary. In fact the severity of powdery mildews is decreased in wet weather or when free moisture covers leaf surfaces. During periods of high rainfall other fungal pathogens become more important, especially the downy mildews.

Fig. 1. White conidia of powdery mildew on poinsettia leaves. *Photo*: Jack Kelly Clark.

Downy mildews are actually foliage blights. Downy mildew diseases are caused by organisms in the class Oomycetes and related to *Pythium* and *Phytophthora*, so as such are not members of the fungal Kingdom but a newly erected Kingdom called the Chromalveolata. All downy mildew species are members of the Peronsosporaceae family and are obligate parasites of higher plants. Downy mildews have complicated and variable lifecycles. They are also compound interest diseases with a sexual stage forming oospores in the leaf cortex that germinate to form sporangia which release zoospores in fallen leaf litter. Infected living plant surfaces also generate sporangiophores that give rise to sporangia and release more zoospores. In some downy mildews, sporangia germinate directly rather than forming a germ tube. Unlike powdery mildews, sporangia form on the underside of leaves, not on upper surfaces. Sporangia formation and disease progression are favored by cool or warm wet conditions but not by hot weather. Free water and high humidity are necessary to the life history of this organism and favor disease development. Recently a new disease caused by *Peronsopora mesembryanthemi* has spread rapidly on *Aptenia cordefolia* or red apple ice plant (fig. 2). Other common ornamental hosts include alyssum, snapdragon and rose. Unlike powdery mildews, downy

Fig. 2. Downy mildew on *Aptenia cordefolia. Photo*: Jose Rodrigues, Waypoint Analytical Labs.

DISEASE FOCUS: Powdery and downy mildews

continued from page 19

mildews invade leaves and stems beyond the epidermis and kill host tissues, often rapidly during wet weather. While early symptoms can look "mildew-like," established downy mildew infections usually lead to necrotic and blighted tissues. Also unlike powdery mildew conidia, downy mildew sporangia are gray-brown in appearance.

Powdery mildews are controlled by a broad range of prophylactic and systemic fungicides. However early applications are advocated because fungicides that eradicate or kill the target fungus will also kill the plant cells it has invaded leading to phytotoxicity as well as control. It is best to control the disease early during spore germination, germ tube elongation or pre-penetration phases of development. Wetting leaf surfaces or increasing leaf wetness can also give some limited control, so inclusion of a wetting agent or surfactant (alone or with a fungicide) can control many of these diseases. Downy mildews are controlled by the same materials that control root rots caused by *Pythium* and *Phytophthora,* so phosphorus acid materials, mefanoxam and other pesticides that control Oomycetes can limit downy mildew formation. Increasing leaf wetness will make downy mildew diseases worse. Generally hot and dry conditions and dry foliage favor control.

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INSECT HOT TOPICS: Coconut rhinoceros beetle

by James A. Bethke

This column focuses on insects that pose a threat to the ornamental plant production industry and have good potential for invasion and establishment in California.

In April, my staff and I attended the Entomological Society of America annual meeting in Honolulu, Hawaii and during our visit it was interesting to note all of the panel traps (fig. 1) in the trees all around the island. More than 2100 panel traps are being used to determine the distribution of the coconut rhinoceros beetle (CRB), *Oryctes rhinoceros* (L.), on Oahu. This beetle is a huge specimen (fig. 2) and the larval stage (fig. 3) principally feeds on coconut and oil palms, but recorded host plants include the date palm and a variety of palms grown as ornamentals such as *Roystonea regia, Livistona chinensis, Corypha umbraculifera, Raphia ruffia* and *Wodyetia bifurcata*. It has also been recorded on pineapple, sugarcane, pandanus and banana. Larvae can be found developing in mulch piles and green waste much like the scarab beetles in Southern California such as the green fruit beetle (fig beetle).

As most of you are probably aware, we have been invaded by several giant palm weevils (red palm weevil and South American palm weevil), but weevils (snout beetles) are very different than the CRB, which is a scarab beetle. Common scarab beetles include June beetle species, dung beetles, rain beetles and chafers, and immatures commonly feed on dung

HOT TOPICS: Coconut rhinoceros beetle

continued from page 20

and detritus (decaying matter). Feeding damage on growing palms resembles the damage caused by the giant palm weevils. CRB damage palms by boring into the center of the crown where they damage and feed on the terminal growing point and on the sap. They damage developing leaves, which eventually expresses as v-shaped cuts in the expanding fronds and holes through the midrib.

Fig. 1. Inspecting a coconut rhinoceros beetle panel trap, Oahu, Hawaii. *Photo*: M. Whitehead, UCCE San Diego.

CRB is a major pest of palms in India, the Philippines, the Palaus, Fiji, Wallis, Nukunono, American Samoa, Samoa (formerly Western Samoa) and Guam.

CRB is native to the Asian tropics, but was accidentally introduced to western and central Pacific islands. Interestingly, CRB was likely introduced throughout the Pacific primarily as a result of the increased sea traffic during World War II (Nishida and Evenhuis 2000). CRB was first detected in Hawaii in December 2013 in Pearl Harbor on coconut trees. Hawaii has begun an intensive eradication effort against CRB. Oahu residents are being asked to check their mulch piles and green waste for any potential breeding populations.

Fig. 3. Adult coconut rhinoceros beetle. *Photo:* Mark Schmaedick, Entomologist, Land Grant Program, American Samoa Community College.

Fig. 3. Coconut rhinoceros beetle, *Oryctes* rhinoceros (L.) larva. *Photo*: Mark Schmaedick, Entomologist, Land Grant Program, American Samoa Community College.

Here in Southern California, we have been faced with the threat of the South American palm weevil, *Rhynchophorus palmarum*, reaching across the Mexican border into San Diego County. This beetle is one of the many giant palm weevils that threaten our palm tree industry in Southern California. CRB is another looming threat that you should be aware of because as we all know, what ends up in Hawaii usually ends up in California.

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HOT TOPICS: Coconut rhinoceros beetle

continued from page 21

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REGIONAL REPORT — UC Cooperative Extension

San Diego/Riverside Counties

Chilli thrips

by James A. Bethke

For years I have been dreading the arrival of the invasive chilli thrips*, Scirtothirps dorsalis* Hood. It's been in Texas and Florida for some time now, and I was hopeful that the bug just didn't like California. Regrettably, it has arrived.

Scientists believe that chilli thrips originated either in Southeast Asia or in the Indian subcontinent, but it is now widely distributed. Chilli thrips was first reported in Florida on October 2, 1991 with subsequent occurrences in 1994, 2004, 2005 and 2007 in various counties of Florida, and it was observed in southeastern Texas in 2005 on landscape roses and on peppers in retail centers. In August of 2015 chilli thrips were found at several residential properties in Orange County, California, and in October, it was detected on roses at the Los Angeles County Arboretum and Botanic Garden. Initially, the pest was rated "Q" and required regulatory action. Thereafter, it was determined that chilli thrips cannot be eradicated, and it is likely to become widespread in California. Recently, it was reduced to a "B" rated pest in California. The B rating still means that some counties may treat this pest as a significant pest, and that it may need local regulation.

Infestations of the invasive chilli thrips poses a number of challenges including:

- Chilli thrips resemble many other thrips species.
- Unlike other thrips, pupae of chilli thrips are generally found on leaves, leaf litter, or on the axils of leaves, in curled leaves, or under the calyxes of flowers and fruits.
- Chilli thrips damage resembles herbicide damage, micronutrient deficiency, or an aphid infestation.
- Insecticide resistance is common.
- Chilli thrips have been implicated in tospovirus transmission.

Chilli thrips are known to infest a wide variety of host plants belonging to more than 200 plant species in 70 plant families, most of them ornamental. It can also attack many trees and tree fruits such as mango, apple, pears, citrus and lychee, and it can attack many field crops such as strawberries, peppers, eggplant, tomatoes, corn and cotton. Infested plants become stunted, and severe infestations can result in total defoliation of the host. Damage to roses includes distorted and elongated foliage, scarred flower buds, and brown, angular spots on the new growth of the roses and defoliation (fig. 1). On many hosts, chilli thrips also start

Fig. 1. Chilli thrips defoliation of roses at the Winter Park Rose Garden in Florida. *Photo*: Lance S. Osborne, University of Florida.

feeding on the upper surface of leaves when the infestation is heavy (fig. 2– 3). Chilli thrips are principally a landscape pest and will damage many of the common landscape plants in California including Indian hawthorne, viburnum, shefflera,

REGIONAL REPORT: San Diego and Riverside Counties

continued from page 23

star jasmine, podocarpus, pittosporum, pyracantha and roses.

If you are facing an infestation, it is recommended that you remove all infested foliage, bag it immediately and place it in the trash. Composting the foliage can lead to a greater level of infestation as the pests leave the infested foliage.

Research suggests that foliar sprays with insecticides containing acephate, imidacloprid, or spinosad are effective for pest control on ornamental landscape plants. Chilli thrips are generally resistant to the pyrethroids such as bifenthrin, cyfluthrin and permethrin, so they are not recommended for control, and are more damaging to beneficial insects.

If you suspect that you have chilli thrips on your plants you are encouraged to seek expert help in identifying the pest. Identifying the pest will help pest control advisors and farm advisors make control recommendations.

Fig. 2. Chilli thrips damage to terminal growth of shefflera. *Photo*: Lance S. Osborne, University of Florida.

Fig. 3. Chilli thrips damage to terminal growth of Indian hawthorn. *Photo*: Lance S. Osborne, University of Florida.

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REGIONAL REPORT: San Diego and Riverside Counties

Continued from page 24

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New Publications from Agriculture and Natural Resources

compiled by Steve Tjosvold

Mealybugs: Pest Notes for Home, Garden, and Landscape

Mealybugs are soft, oval, wax-covered insects that feed on many plants. Usually found in colonies, they are piercing-sucking insects closely related to soft scales but lack the scale covers. This free publication describes the life cycle and plant damage associated with mealybugs. Although the control methods are targeted for home gardeners and landscapers, growers in commercial greenhouse and nursery operations will find the photos and descriptions of specific mealybug species commonly found in California helpful in pest identification.

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