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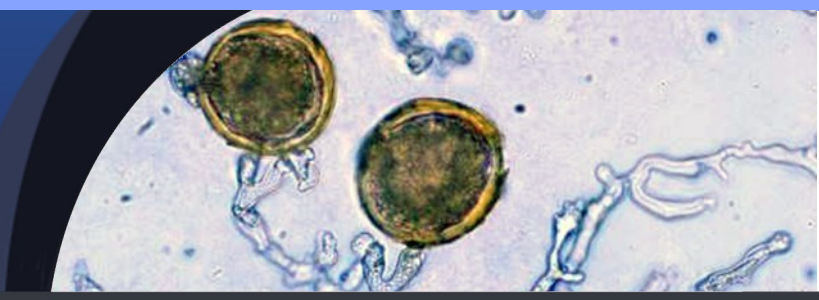

Inside

Phytophthora in Restoration and Forest Nurseries

***by* Laura Sims *and* Matteo Garbelotto**

Diseases caused by *Phytophthora* are one of the most important problems for plant health in wildland and forest areas, especially following a rainy year. Specialized nurseries that grow plants for restoration and forest plantings play an important role in supplying healthy plants for these restoration and forest projects. Diseased nursery plants and infested potting soil with *Phytophthora* pathogens can be moved into wildland and forest areas and can subsequently become a source of new infections of other plant hosts. The long-term cost of managing diseases after these pathogens are introduced are much greater than preventing plants from becoming infected in the nurseries in the first place. Thus reducing *Phytophthora* in nurseries is a key strategy for protecting the health of wildlands, especially for those nurseries producing native plants.

Without management and with conditions supporting disease, a high percentage of nursery plants could be a source of *Phytophthora* disease inoculum, as demonstrated in a survey conducted in Northern California nurseries where an average of 44% of native woody perennial plant crops were found to carry infections (Sims et al. 2017). These infections can potentially spread when outplanted to other sites,

Editor's Note

The heavy winter storms that persisted this past winter and spring brought welcome rains that filled reservoirs and aquifers. However, these conditions also optimized conditions for the "water mold" *Phytophthora* that is capable of rotting roots and killing many types of ornamentals and native plants. *Phytophthora* includes several species that are typically often found in nurseries and two new exotic species, *P. ramorum* and *P. tentaculata.* All species are capable of causing widespread damage, especially if diseased plants are outplanted in wildland and forest areas. Three feature articles are included in this issue lead by Sims, Tjosvold and Downer on various aspects of *Phytophthora* and its control, as well as management of other root diseases. Moreover, a new disease, boxwood blight, has been discovered in Bay Area landscapes and is described in Tjosvold's Regional Report.

♦**Steve Tjosvold and Julie Newman**

Phytophtora in Restoration and Forest Nurseries

continued from page 1

putting neighboring landscapes and plant communities at risk. Due to the lasting environmental and economic impacts these pathogens cause, many managers growing native plants in restoration nurseries are working on preventing *Phytophthora* diseases by implementing best management practices (Sims et al. 2016a). Managers of other types of nurseries are also encouraged to improve their practices for growing healthier plants. Preventing *Phytophthora* in the nursery not only protects landscapes and surrounding areas from disease when plants from the nursery are outplanted, it can also drastically reduce costs associated with fungicide use, reduce plant loss from disease, and can help nurseries meet the increasing standards of homeowners and agencies. Outlined herein is information on what *Phytophthora* is, prevention and management strategies to reduce the risk of *Phytophthora* in nursery stock, and some online resources.

What is Phytophthora?

Phytophthoras are worldwide-recognized plant pathogenic microorganisms that cause root and collar rot diseases of herbaceous and woody plants (Erwin and Ribeiro 1996). Additionally, some *Phytophthoras* infect aboveground plant parts and cause aerial diseases. Due to their ability to produce swimming spores and the importance of water in their lifecycle, *Phytophthoras* are often referred to as water molds: waterborne, these pathogens move and spread through infested water. They are also soilborne, spreading by the movement of infested soil, including soil stuck to tools, containers, or shoes. In addition to producing swimming spores, many *Phytophthora* species also produce thick-walled survival spores that can make disease eradication difficult once introduced to wildlands and urban planting areas. For example, chlamydospores are a distinctive feature of P. *ramorum* (see UCNFA banner on top of previous page for a photo of the chlamydospores by David Rizzo, Department of Plant Pathology, UC Davis). They form within colonized leaves and can persist in soil and leaf litter.

Phytophthora infection begins with the release of swimming spores that move through the water. Root-rotting *Phytophthora* species, in particular, are attracted to plant root exudates. Once susceptible plant roots are infected, root-rotting *Phytophthora* species can degrade roots and cause disease that can spread upward into the root collar and sometimes the stems. Stem disease and infection of leaves (in *Phytophthoras* that cause aerial disease) can also be caused by direct contact, for example following water movement or splashing of infested water or soil. The sporangia or zoospores of aerial *Phytophthora* species may spread short distances in air currents or with wind-driven rain. Root-rotting *Phytophthora* species on the other hand, mainly spread through soil and water movement only. *Phytophthora* species cannot be seen with the naked eye unless grown in culture in a laboratory. However, if fungicides do not suppress *Phytophthora* disease development, it is often possible to see visible symptoms on host plants (fig. 1). These symptoms are described in detail in our next feature article in this newsletter.

Fig. 1. *Ceanothus thyrisflorus* infested with *Phytophthora* sp. pathogens in a restoration nursery. *Photo*: Laura Sims.

There are over 100 described species in the genus *Phytophthora* and several of these can be a problem in nurseries. Additionally, hybrids and two new exotic species, *P. ramorum* and *P. tentaculata*, have been detected in nurseries, and these are especially of concern should they be introduced and spread to urban landscapes and wildlands.

Hybrid species may have an improved ability to cause disease when compared to parent type *Phytophthora* species. Nurseries provide an environment in which otherwise ecologically distant species that may be closely related have a chance to meet and reproduce.

P. ramorum, which causes sudden oak death, has already caused widespread mortality in native oaks and tanoaks in coastal areas of central and northern California and southwestern Oregon. First officially detected on rhododendrons in a Santa Cruz County nursery, it has since spread to 14 counties in California; it has also been detected in many other states and in British Columbia, and is widespread in Euro-

Phytophtora in Restoration and Forest Nurseries

continued from page 2

pean nurseries. (Tjosvold et al. 2005; Tjosvold 2015).

P. tentaculata has recently been detected in several California native plant nurseries and restoration sites, and these are the first detections of *P. tentaculata* in the United States (Rooney -Latham and Blomquist 2014). *P. tentaculata* can cause root-rots, collar cankers, and stem cankers, which may result in the death of infected plants (Sims et al. 2016b), but it is unlikely to spread aerially as *P. ramorum* does. Managers of native and restoration nurseries must be on the lookout for these pathogens and implement best management practices to prevent the spread of new diseases.

Prevention and Management

Keep *Phytophthora* out of your nursery from start to finish by starting with healthy plant materials and maintaining sanitary conditions throughout the growing process. Start with healthy plant materials by using clean propagation practices and ensuring that plant material brought in from other nurseries is clean (Griesbach et al. 2012). Then, prevent plant exposure to *Phytophthora* during production phases by growing plants in clean media and containers, preferably on raised benches. Make sure to remove diseased crops. Only use clean tools and maintain proper irrigation practices throughout the life of the plant. If the nursery is involved in transporting nursery plants to the planting destination or to other nurseries, ensure that plants are moved without being exposed to pathogens in the process (sanitary truck bed).

Propagation. Seed is the preferred source of propagation material. This is because *Phytophthora* pathogens are often not transmitted by seed, so disease problems may be prevented if seeds are from a clean and healthy plant source. Avoid collecting seeds from the ground. Seeds that have soil or debris on them or are from an area where they could be immersed in site water could be contaminated with *Phytophthora.* If seeds must be collected following dissemination from the plant, then lay down a clean tarp, allow the plant to disperse seeds onto the tarp and retrieve seeds from the tarp only. Also, avoid collecting seeds from plants that appear unhealthy and use only a healthy source plant if it is necessary to propagate from cuttings. Treat low-growing seed collections and all cuttings with a fungicide dip following label protocol. All chemicals should only be applied following safe handling guidelines that are required by law and allow workers to use chemicals safely (CDPR handout 2016); it is always important to inform workers about the safe handling of chemicals and to post the safety data sheets (SDS) for the ones in use. It is of particular importance to keep propagation areas clean and sanitary and to do so safely. Use only clean (sanitized) tools and containers and pathogen-free root media in the propagation area. Organize your nursery so that propagation areas are away from potential sources of contamination.

Sanitation. All potting mix should be stored in original bags or in sanitized, covered containers until use. Reuse of media increases the potential of disease. Ground beds and potting mix that is reused should be treated. Even some new mixes, notably those containing peat, may benefit from treatment prior to use (Mathews et al. 2014). UC IPM recommendations for managing *Phytophthora* are to steam (at 140°F for 30 minutes), solarize (double-tent at 160°F for 30 minutes or 140°F for 1 hour), or chemically treat growing media before use (Koike et al. 2009). Composting alone is not sufficient for killing *Phytophthora*, as is difficult to reach adequate temperatures evenly and for long enough to kill the pathogen in these systems.

Used containers should always be in good condition, cleaned, and sanitized before entering your nursery system. Proper disinfection of containers to eliminate *Phytophthora* and other pathogens (as well as killing most pathogens, pests and weed seeds) can be the difference between success and failure of containerized plants. First, wash containers to remove soil and debris. Then, disinfect containers by soaking in a 10% bleach solution for 30 minutes. (A 10% bleach solution can be made by adding one part standard household bleach containing 5 to 6% sodium hypochlorite to nine parts water.) Alternately, use aerated steam to disinfect containers by heating them to 180° F for 30 minutes (test container materials first to determine if they can withstand the heating process). Another option is to immerse washed containers in an 180° F hot water bath for 30 minutes.

Assume the ground in the nursery site is contaminated with *Phytophthora* and other pathogens. Avoid contamination from soil and debris by keeping plants and propagation materials elevated on an open-mesh bench, 3 to 4 feet above the ground, and discard any plants placed directly on the ground. Hang up hoses after use; do not let the hose nozzle or spray wand contact the soil to avoid transferring pathogens from the ground to growing areas. If the hose nozzle or spray

Phytophthora in Restoration and Forest Nurseries

continued from page 3

wand is on the ground or becomes soiled, wipe it clean of debris and apply a disinfectant before using.

Collect, bag and remove crop residues; leaf debris; pruned plant material; and unhealthy and unmarketable plants in production areas on a regular basis. Place in a covered dumpster away from and downwind of healthy plants and production areas. Disinfect propagation areas, greenhouses and shadehouses after every crop rotation. Use a power washer to remove all visible soil and debris, then spray surfaces with a disinfectant active against *Phytophthora,* such as alcohol (70%) or quaternary ammonium.

Clean and sanitize tools before and after use, using sets of tools designated for particular tasks. This can help to keep any breaches localized, making any necessary remediation simpler.

Exclusion/quarantine: When purchasing plant material from other nurseries, source from nurseries following best management practices for preventing *Phytophthora* and inspect incoming shipments to make sure plants are free from disease symptoms. One way to exclude *Phytophthora* is to quarantine new plants before incorporating them into the production system by placing them in a separate greenhouse or an isolated nursery area. (Exclusion is a mechanical control technique that consists of using barriers to prevent new pests and pathogens from entering an area.) Give the quarantined plants time (8 weeks) to develop symptoms in case fungicides or past conditions are inhibiting symptom development. If symptoms develop on plants in the quarantined area, dispose of the plant, soil and containers according to disposal guidelines for your area (e.g., there are specific CDFA disposal guidelines to be followed for *Phytophthora ramorum*); clean and sanitize benches.

Proper irrigation: Appropriate irrigation is extremely important for managing plant health and reducing the movement of *Phytophthora* from adjacent or nearby diseased areas in water. Practical strategies to improve irrigation include grouping plants in the nursery based on their watering needs, carefully watering to avoid splashing soil and water, and monitoring soil moisture levels. Start watering early (such as before dawn from 2 to 3 a.m.) to avoid prolonged leaf wetness and avoid overhead irrigation. Do not place emitters so that water sprays directly onto stems or foliage. Do not irrigate areas that are already wet because waterlogged soil can accelerate disease development. Also, do not stress plants by underwatering as this can predispose plants to disease. Instead, conserve water and promote plant health by scheduling irrigation based on local evapotranspiration or by installing soil moisture probes or tensiometers.

Provide regular maintenance of the irrigation system to maintain proper pressure; replace irrigation emitters and repair broken systems. Monitor irrigation water from any source other than a municipal water supply and test for waterborne pathogens at least bi-annually. Testing should be to confirm that the water is free from pathogens before use on plants or planting media.

Remediation. Plants that are exhibiting symptoms such as stunted growth and chlorotic foliage should be sent to a professional plant pathology laboratory for diagnosis, or in-field tests kits can be used. See our next feature article for a detailed discussion about plant symptoms and the identification procedures for *Phytophthora*.

Discard unhealthy plants that are *Phytophthora*-infected, following local guidelines for disposal. Clean and sanitize the area where diseased plants are detected. Replace material beneath benches if they become contaminated.

In ornamental nurseries, economic losses due to *Phytophthora* are significant, and disease management typically includes the considerable use of fungicides. Plants in areas surrounding locations where pathogens have been confirmed are usually protected by using registered fungicides that are active on *Phytophthora* species (see UC IPM Guidelines listed in references). However, because fungicides suppress disease symptoms, chemical treatment can make early detection of the pathogen in monitoring programs more difficult, and cause detection problems. Moreover, concern has been raised that the use of fungicides in restoration and forest nurseries can result in infected plants that show no obvious symptoms being planted in wildlands. Then, as disease suppression declines and the chemicals degrade, the pathogens can resume activity, leading to both plant decline and infestation of the planting site (Working Group for Phytophthoras in Native Habitats 2016).

Phytophthora in Restoration and Forest Nurseries

continued from page 4

Online Resources

Two online resources were recently developed for managing *Phytophthora* in restoration, forest and native plant nurseries: *Presidio Phytophthora Management Recommendations* and *Guidelines to Minimize Phytophthora Pathogens in Restoration Nurseries*. Both online resources are included in the references below, along with other helpful resources (e.g., CalPhytos.org and UC Berkeley Forest Pathology Lab websites). It is recommended that growers consult hese resources for more in-depth management guidelines. Additionally, the management practices and recommendations that are listed in these references can be used to design and execute a best management plan to meet the needs of your particular nursery and help you avoid future outbreaks of *Phytophthora*.

L. Sims is Postdoctoral Researcher in the Department of Environmental Science, Policy, and Management, UC Berkeley; M. Garbelotto is Extension Specialist in Forest Pathology and Adjunct Professor in the Department of Environmental Science, Policy and Management Department, UC Berkeley.

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Phytophthora Crown and Root Rot Symptoms and Detection

by **Steve Tjosvold, Laura Sims *and* Matteo Garbelotto**

What do you look for when scouting nursery plants for Phytophthora root rot? How do you know your nursery plants have *Phytophthora* disease? In the accompanying newsletter article "*Phytophthora* in Restoration and Native Plant Nurseries," Laura Sims and Matteo Garbelotto describe what *Phytophthora* is and how to manage it in nurseries. But here is a description of symptoms and a pictorial guide to help you home in on plants that may have Phytophthora root diseases in your nursery.

The expression and severity of Phytophthora crown and root rot depend on the particular plant host, *Phytophthora* species and environmental conditions. Some plant species limit infection and colonization of their root system resulting in the loss of just a small number of feeder roots. Limited colonization may only cause mild or even no apparent aboveground symptoms. There may be some limited plant stunting or leaf chlorosis evident when compared to healthy plants. However, there is not always a straight-line relationship between the aboveground symptoms (see fig. 1,4,6,8 for examples) and belowground infections (see fig. 2,3,5,7,9).

Fig. 1. *Frangula californica* (California coffeeberry) with varying degrees of Phytophthora root rot (infected with *Phytophthora multivora*). *Photo:* S. Tjosvold.

Sometimes infected plants may display limited aboveground symptoms and actually the roots are quite infected. If unmanaged, these plants can act as cryptic sources of the pathogen and its infectious spores can spread to other plants.

Fig. 2. *Frangula californica* (California coffeeberry) root systems are uniquely yellow to orange when healthy. *Photo:* S. Tjosvold.

Usually, however, when root rots are more moderate or severe, aboveground symptoms can include obvious stunting and shoot dieback. Leaves can be smaller than normal and have chlorosis or interveinal chlorosis.

Wilting can occur even with adequate soil moisture. When *Phytophthora* infects at or develops into the root crown near the soil line, the disease is described as a "crown rot."

Fig. 3. *Frangula californica* (California coffeeberry) with necrotic roots infected with *Phytophthora multivora*. *Photo:* S. Tjosvold.

Often at this stage, leaves may droop, and the plant dies. Cutting just under the bark at or just above the soil line may reveal dead inner bark tissue.

The dead tissue may be reddish brown, brown, or black and will differ from healthy

Phytophthora Crown and Root Rot Symptoms and Detection

continued from page 6

tissue, which can be white, green, or pink depending on the type of plant or tissue. Of course, knowing what a healthy plant of the particular species of concern should look like will help in the diagnosis. Root balls must be examined by carefully removing the pot to expose the roots. Sometimes gently shaking, or washing the soil mix from the outer portion of the root ball, can allow for a better examination.

Diseased roots can be reddish brown to dark brown while

Fig. 4. *Diplacus aurantiacus* 'Trish' with various degrees of Phytophthora root rot (infected with *Phytophthora cryptogea). Photo:* S. Tjosvold.

healthy roots are often white to tan, depending on the plant species. So, just as it is important to know what the aboveground parts of healthy plants look like so that you can compare with unhealthy plants, it is also important to know what healthy roots look like in your species for comparison.

Do not get fooled by healthy-looking roots of weeds that might infiltrate the root ball. Feeder roots can be rotted away, and heavier roots can be discolored. Fleshy roots of some plant species can be brown, water-soaked and flaccid. They may also be brittle, thin and rotted inside, while healthy roots are often turgid and crisp.

All or some of these described symptoms might be caused by other root pathogens such as *Pythium*, *Rhizoctonia*, *Thielaviopsis*, *Fusarium* or *Cylindrocladium*. Abiotic problems such as flooding, drought, extremes of heat or cold, excess fertilizers, or toxic level of salts in irrigation water might also cause these symptoms. Identifying *Phytophthora* quickly in the field can be important to make timely management decisions.

Agdia (Elkhart, IN) supply simple immunological test kits that detect *Phytophthora* species in minutes (fig. 10). A positive detection can usually help with the diagnostic process and get you quickly on the path to managing the problem.

Fig. 5. *Diplacus aurantiacus* (sticky monkeyflower) with Phytophthora root rot (infected with *Phytophthora cryptogea*). All fine feeder roots are infected and necrotic. *Phytophthora* has not killed larger roots yet. *Photo*: S. Tjosvold.

Sometimes, however, a positive result might be deceiving because these tests are not entirely accurate. Sometimes they can react to certain *Pythium* or *Phytopythium* species. These latter two groups of species may or may not be the primary cause of root disease or even a problem at all.

Some of these are soil microbes that only break down already dead material, and

Fig. 6. *Arctostaphylos uva-ursi* with various degrees of aboveground symptoms; all have Phytophthora root rot (infected with several *Phytophthora* species). *Photo*: S. Tjosvold.

Phytophthora Crown and Root Symptoms and Detection

continued from page 7

many we do not fully understand yet. A non-positive reaction of the immunological test might be deceiving also.

Sometimes the tested root tissue may not have been sampled from infected root pieces, and certain *Phytophthora* species do not react with the tests.

The reason why these problems can arise is that the tests were developed for particular *Phytophthora* species that occur on leaves. It just so happens to work pretty darn good on roots as well, but not wholly without issues.

Fig. 9. *Diplacus aurantiacus* (sticky monkeyflower) with Pythium root disease symptoms (infected with *Pythium cryptoirregulare*). Note the many light-brown water-soaked root regions and that this plant is also root bound. Poor root and overly wet conditions may have contributed to the unhealthy nature of this particular plant. *Photo*: S. Tjosvold.

Fig. 7. *Below left*: *Arctostaphylos uva-ursi* with Phytophthora root rot on feeder roots primarily in the lower soil profile. *Photo*: S. Tjosvold.

Fig. 8. *Top right: Diplacus aurantiacus* (sticky monkeyflower) with Pythium root rot (infected with *Pythium cryptoirregulare*). *Photo*: S. Tjosvold.

For these reasons, to gain identification for critical management decisions, it would be advisable to take disease samples to a professional plant pathology lab where cultures can be obtained and identified.

Fig. 10. Immunostrip test for *Phytophthora*. Symptomatic roots are sampled, macerated in a buffer test solution and pouch. The test strips inserted in the pouch indicate results in as little as few minutes. Test strips have a magenta-pink-colored "control line" indicating a valid test, as seen in both test strips here. If *Phytophthora* is detected, then two lines appear: a control line and a "*Phytophthora*-discovered" line. You can see the latter line next to the red arrow on the strip to the left (although more weakly) but not on the strip to the right. *Photo*: S. Tjosvold.

S.Tjosvold is Environmental Horticulture Advisor Farm Advisor, UC Cooperative Extension, Santa Cruz and Monterey counties; L. Sims is Postdoctoral Researcher in the Department of Environmental Science, Policy and Management, UC Berkeley; M. Garbelotto is Extension Specialist in Forest Pathology and Adjunct Professor in the Department of Environmental Science, Policy and Management Department, UC Berkeley.

Can Fresh Wood Chip Amendments Suppress Root Pathogens?

***by* Jim Downer**

Plant pathogens, especially root pathogens, survive in container media and can be some of the most destructive nursery pests. Root pathogens are successful because they are hidden; symptoms don't show on foliar parts until significant percentages of roots are lost (fig. 1). Often more than 50% of absorptive roots are dead before wilt, die back, color changes, or leaf drop are apparent. Early symptoms of root disease such as growth cessation or stunting are usually not observed. Because root-compromised plants use less water due to loss of function, media become and stay wet or saturated. This increases media breakdown and the development of anaerobic chemistries in the bottom of the container. These conditions prevent root regeneration, dooming plants to early failure.

Fig. 1. Incipient root rot does not show on foliage but roots may be discolored. *Photo*: J. Downer.

In a series of articles in *Greenhouse Grower*, Brian Jackson and other researchers at North Carolina State University (NC State) detail the benefits of using wood fiber and other wood components in container media, which include increased root growth in floricultural crops (Jackson 2016; Owen and Jackson 2014, Owen and others 2014). Most of the NC State research has focused on small pine wood chips — fresh not composted — engineered as an aggregate for use in peat-based media.

While non-composted redwood and fir bark may break down in container media and eventually decrease porosity, Jackson's research showed that amending peat-based media with the specific fresh pine chips studied at NC State increased porosity, making it a good substitute for perlite.

In addition to vigorous root growth and increased media porosity, fresh wood chip amendments may suppress some soilborne diseases. Jackson's popular articles do not mention this potential benefit, but there is some evidence that supports this assumption presented in another NC State paper (Kaderabek and others 2013). In this study, disease severity of *Pythium* and *Rhizoctonia* was often less in peat-based media amended with fresh wood chips than in peat-based media alone; the media without fresh wood chips also had greater disease earlier in the cropping cycle, which could indicate that the wood chips somehow suppressed disease. The increased porosity in the media with fresh wood chips could explain the observed reduction in disease severity. For example, Filmer and others (1986) showed that increases in porosity of container media lead to control of Phytophthora root rot of toyon. I like to call managing diseases by modifying media components substrate-mediated disease control (SMDC). Alternatively, since fresh pine bark are known to contain inhibitors that may suppress some soilborne diseases (Hoitink and Fahy 1986), it's possible that the fresh wood chips used in the study also contained inhibitors. Moreover, fresh wood chip amendments could potentially suppress soilborne diseases by enhancing biological control of root pathogens.

Soilborne plant pathogens can be parasitized by other non-pathogenic or saprophytic fungi resulting in disease suppression (Hoitink and Fahy 1986). A typical example is the hyperparasitism of *Pythium* by *Trichoderma*. The work by Gravel and others (2009), in which *T. harzianum* (applied as the product Rootshield) successfully limited geranium root disease in peat-based media, is typical. However biological controls, while effective experimentally, often fail in sustained production and as plants grow to larger sizes.

Trichoderma is a wood-inhabiting fungus and preferentially grows and sporulates on fresh wood (fig 2). In my own experience and in observations from experiments that I conducted with other researchers, media lacking fresh wood chips may be less likely to sustain *Trichoderma*. For example, we observed increases of *Trichoderma* on fresh wood chips associated with concomitant loss of viability of *Armillaria mellea* inoculum incubated in piles of fresh yardwaste over time (Downer and others 2008).

Can Fresh Wood Chip Amendments Suppress Root Pathogens?

continued from Page 9

Targeted or classical biological control seeks to "inoculate" or "inundate" the environment with parasitic or predatory organisms respectively. Curiously SMDC does not require inundative releases of biological control agents. If the substrate for microbial growth is in place, biocontrol fungi can develop on those particles and impart their disease suppression to the media. For this reason, media made of stable materials such as pine bark or peat moss may be less likely to support growth of biological control agents than more labile components such as fresh wood chips.

Fig. 2. Fresh wood chips become infected with *Trichoderma* spp. over time. Photo: J. Downer.

If growers decide to use wood chips in container media, one caveat is that nitrogen be supplied in quantities to support both microbial and plant growth. Fresh wood chips added to media increases the carbon to nitrogen ratio of the medium and could lead to nitrogen depletion, depending on the size of the wood chips. See the article published by Owen and others (2014) as part of the *Greenhouse Grower* series, or their publication in HortScience (2016), for details concerning fertilization with pine wood chips. When the specific pine chips studied at NC State were used in peat-based media at rates of 10% to 30%, no differences were found in nitrogen use (100 to 200 ppm nitrogen allowed for normal growth of zinnia and marigold), liming practices, or plant growth regulator efficacy/response compared to the same rates of perlite.

Some knowledge is available from published scientific studies on beneficial organisms (antagonists) involved in suppression of plant pathogens in media amended with composts, including those with wood components. For example, *Trichoderma* spp. and *Gliocladium virens* are among the most abundant fungal taxa associated with suppressiveness in composted hardwood bark (Hoitink and Fahy 1986). Unfortunately, biocontrol organisms associated with fresh wood fiber and other wood components in container media is little studied and there are few references to it in the literature (Brian Jackson, personnel communication, 2017). Research on SMDC and the ability of non-composted wood amendments to regulate the type and quantity of biological control organisms in container media is needed.

Jim Downer is Environmental Horticulture Farm Advisor, UC Cooperative Extension, Ventura County

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Can Fresh Wood Chip Amendments Suppress Root Pathogens?

continued from page 10

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GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

***by* Donald J. Merhaut**

The rooting media in container production is influenced by three main factors: substrate types, water quality and temperature. There are two main variables, pH and electrical conductivity (EC), which can be measured to indirectly determine nutritional status of the rooting media and impacts on production from water sources. How to measure pH and how it relates production processes is the subject for another newsletter. In the following article, I will focus on the EC only, what it means to your production, how to properly measure EC and what action to take if EC levels are high

Irrigation Water Electrical Conductivity (EC)

The electrical conductivity (EC) is a measure of the ability of water to conduct electricity. Different laboratories may use different units of measure. These units and their conversions are given in table 1.

Table 1. Common units of measure for electrical conductivity and their conversions.

Abbreviation	Units of Measure
dS/m	Decisieman per meter
mS/cm	Millisieman per centimeter
µS/cm	Microsieman per centimeter
mmho/cm	Millimho per centimeter
µmho/cm	Micromho per center
ppm	Parts per million

Conversions: 1 dS/m = 1 mS/cm = 1 mho/cm = 1000 µmho/cm = 1000 µS/cm = 700 ppm

GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

Continue from page 11

Deionized water cannot conduct an electrical current because all the ions have been removed. The EC of water increases when positively and negatively charged ions, such as sodium (Na^+), fluoride (F^-), calcium (Ca^{2+}), sulfate (SO_4^{2-}), nitrate (NO_3^-) and ammonium (NH_4^+), are added to the water. Potable water has some salts, such as F^-, Na^+ and NO_3^-, which will increase the EC, usually in the range of 0.5 to 0.8 dS/m. For other water sources, such as well water, the EC may be elevated due to the presence of carbonates (CO_3^{2-}) and bicarbonates (HCO_3^-), the presence of which also contributes to alkalinity of the water. Alkalinity is the measure of water's ability to increase rooting media pH and will be addressed in a future newsletter. In brief, there is no direct correlation between alkalinity and EC of water sources, so EC measurements cannot be used to estimate water alkalinity or the functionality of acidification processes to reduce alkalinity (see example in table 2).

Table 2. The effects of acidification to reduce alkalinity on irrigation water. Even though pH and alkalinity decreased with acidification, there was no effect on electrical conductivity.

Location	Alkalinity ($CaCO_3$ meq/L)	Electrical Conductivity (dS/m)	pH
Florida – acidified well water to remove 80% alkalinity	1.0	0.9	4.8
Florida – acidified well water to remove 40% alkalinity	3.0	0.9	6.4
Florida – untreated well water	5.0	0.8	7.4

From Albano, et al. 2017. Irrigation water acidification to neutralize alkalinity for nursery crop production: substrate pH, electrical conductivity, and nutrient concentrations; and plant nutrition and growth. HortScience. In Press.

Other sources which may increase well water EC include salt water intrusion or other dissolved minerals. If a water source has a relatively high EC (> 1.0 dS/cm), it may cause production problems, especially if fertilizer, which will increase EC, is injected into the irrigation system.

GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

Continued from Page 12

Electrical Conductivity of Irrigation Water Fortified with Fertilizer

When injecting fertilizer into the irrigation system, measuring EC can provide an easy way of monitoring that fertigation system to ensure that it is functioning properly. There are a few rules to follow when utilizing EC to estimate proper fertilizer injection:

1. Keep a log book of EC readings.
2. Let the irrigation system run before taking a sample.
3. If water sources change, the EC of the new water source needs to be considered.
4. If fertilizer formulations change, new EC readings need to be established, as different fertilizer types can change the EC, even if the nitrogen rates are similar (table 3).

Table 3. The electrical conductivity of fertigation water containing different fertilizer types.[*]

Fertilizer Name	Chemical Formula	Concentration (ppm)	Electrical Conductivity (dS/m)
Potassium Nitrate	KNO_3	100 ppm nitrogen	0.76
Ammonium Nitrate	NH_4NO_3	100 ppm nitrogen	0.40
Ammonium Sulfate	$(NH_4)_2SO_4$	100 ppm nitrogen	0.77
Ammonium Chloride	NH_4Cl	100 ppm nitrogen	0.78
Diammonium Phosphate	$(NH_4)_2HPO_4$	100 ppm nitrogen	0.63
Urea[†]	CH_4N_2O	100 ppm nitrogen	0.03
Urea[†]	CH_4N_2O	500 ppm nitrogen	0.05
Ferric Sodium Ethylenediaminetetra acetic acid (FeNaEDTA) chelate	$C_{10}H_{12}N_2O_8FeNa$	50 ppm iron	0.05

[*]In this example, deionized water was used, which has an EC of 0.00 dS/m. All chemicals used were reagent grade. Commercial fertilizers may result in a higher EC due to impurities.

[†]While urea has an EC of near 0.0 dS/m, in production systems it can quickly break down to ammonium (NH_4^+) and ammonia (NH_3), which can be rapidly taken up by plants and can be toxic in high rates. This is especially a problem during high summer temperatures when this chemical conversion is faster.

GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

continued from Page 13

There are two major "take home" messages from the fertilizer examples shown in table 3.

1. Fertilizer source can have a large impact on EC, even if nutrient concentrations are similar. Even though there are six different nitrogen sources in this example, all containing 100 ppm nitrogen, the EC ranged from 0.03 (urea) to 0.78 (ammonium chloride).

2. Only compounds that carry a positive or negative charge or dissociate (break apart) into ions in water will increase EC. Unlike molecules such as ammonium chloride (NH_4Cl), which break apart into the charged ions of NH_4^+ and Cl^-, urea does not dissociate ("break up") into charged molecules when dissolved in water. Therefore, urea, an uncharged molecule, does not contribute to the EC of the water. Thus, even at 500 ppm nitrogen from urea, the EC is still near 0.0 dS/m. Note that in the soil, urea eventually will break down into ammonia and ammonium.

3. Similar to urea, chelates are also uncharged molecules, so the contribution to the EC is negligible, as seen with this iron chelate example.

Monitoring Rooting Media EC

While it is critical to monitor EC in the irrigation water sources and fertilizer injection systems properly, it is also important to monitor rooting media EC. Some commercial rooting media contain fertilizer or you may add granular fertilizer types into the media as part of your production programs. In addition, as organic components break down, other salts may be released from the media. Over time there can be a buildup of salts in the container, and in some cases this can be severe (fig. 1). In normal production practices, it is usually recommended to have a leaching fraction of 25%, so if you add 1 liter of water to a container, 0.250 liter drains from the container bottom. By conducting the following "PourThru Method" (see below), one can monitor salt buildup through EC measurements and changes in rooting media pH with a pH meter. Both EC and pH meters are available in portable pen-sized electrodes, making it much easier to perform on site measurements.

Fig. 1. Salt buildup up in a container receiving overhead irrigation with secondary water sources such as reclaimed municipal water or water sources high in alkalinity. Salts accumulate where wicking of moisture occurs from the media. In this example, evaporation occurs from the top of the container and on the inside on the southwestern side of the container (not shown). This will result in root dieback in these regions. *Photo*: D. Merhaut.

Instructions for Monitoring EC and pH in Container Media

This method is adapted from Cavins et al., 2000, Monitoring and managing pH and EC using the PourThru Extraction Method, NC State University Leaflet 590 (https://content.ces.ncsu.edu/monitoring-and-managing-ph-and-ec-using-the-pourthru-extraction-method):

1. Irrigate containers as you would do in your standard production practice, using the water source you usually use, even if you inject with fertilizer. This can be drip, overhead irrigation, or hand watering. The container should be thoroughly saturated. If the substrate contains a high percentage of peat moss or other substrate which is hydrophobic (does not rewet easily when dry), use other containers that do not have this problem.
2. Allow containers to sit approximately one hour for drainage to occur.

GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

continued from Page 14

3. Place the container in a bucket or saucer. A small paint bucket works well for #1 container (fig. 2 and 3). It is also recommended to place an inert object such as a PVC ring in the container to elevate the pot above the bottom of the collection bucket. This insures that all solution drains from the container and prevents pots from sitting in the drainage water.
4. Add only enough distilled water so that you can collect about 50 millilters of leachate from the pot. It is preferred to use distilled water since this will have an EC value of 0.0 dS/m. However, potable water may be used, but subtract out the EC contributed by potable water. As a side note, if the potable water has high alkalinity, it will interfere with pH readings. The important part of this step is to not add too much water, because this will dilute the sample, causing erroneously low EC values.
5. Allow the pots to drain into the saucer or collection bucket, approximately one hour, depending on media type and size of containers.
6. Measure the EC with a portable EC meter, following the instructions for the specific meter that you are using.

Fig. 2. *Below right:* A 2-L paint bucket with a 3-inch PVC ring allows a #1 pot to be placed inside and elevated for use in collecting leachate.

Fig. 3. *Top left:* A mother-in-law's tongue (*Sansevieria*) in a #1 pot placed inside a paint bucket to collect leachate.

Some key notes regarding the PourThru methods:

1. Perform a few test runs to determine the amount of water needed to collect approximately 50 milliliters of leachate. This will vary with container size and type of rooting media.
2. Perform the method on at least three to five pots and get the average reading for those containers.
3. Keep the containers level so that the water flows through the media uniformly.
4. Choose plants for the test carefully. The growing media conditions from container to container throughout a production block can be quite variable. For example, containers that are on the outside of the southwest side of the production bed tend to dry out faster and will give higher EC readings. The PourThru method can be used to see if excess salt accumulation is indeed occurring on these production bed perimeters.

GET CULTURED: Monitoring electrical conductivity of irrigation water and rooting media

continued from Page 15

5. Be consistent with sampling procedures each time the test is conducted. It is recommended to also record the volume of leachate collected. This way, you can tell if testing procedures are uniform, especially if different individuals conduct the tests. If you decide to weigh the leachate rather than measure the volume in a beaker, 1 milliliters of water weighs 1 gram.
6. In the summer, you may need to do tests every month or every other week. Also, more frequent testing will be necessary if plants show signs of salt stress, such as leaf necrosis on the outer edges of older leaves or plant wilting even after an irrigation event.
7. Completely rinse and dry the paint buckets or leachate collection trays. Any algae buildup and dirt can give erroneous readings.
8. Keep records of the EC measurements, current fertilizer programs (especially when brands of fertilizer change or when fertilizer stocks are made), weather conditions and canning date of production block.
9. If media shrinkage is a problem, keep records of levels of rooting media in containers. If media rooting media levels have decreased significantly, it may be an indication of incomplete composting of organic materials.

Once the readings have been determined, the EC range will indicate what, if any, action is required to improve the production process. Some ranges of EC are listed in table 4. Most propagation programs perform better under EC values of less than 1.5 dS/m since new roots of young plants can be particularly sensitive to high salts. The medium range of EC is usually recommended for crops with low fertilizer requirements such as camellia, azalea, ferns and blueberries. The high EC values usually are recommended for woody perennials with higher fertility requirements. If readings are above the ranges listed, it may be necessary to leach containers with irrigation water to remove excess salts. Other options are to decrease or turnoff fertilizer injection for several days.

Table 4. General guidelines for electrical conductivity utilizing a PourThru method in containerized crops. In conjunction with these guidelines, observe plant performance: necrosis on the outer margins of older leaves and plant wilting, even after an irrigation event may be a sign of salt accumulation in the rooting media.

Low EC 0.0-1.5 dS/m	Medium EC 1.5-2.5 dS/m	High EC 2.5 -3.5 dS/m
Propagation beds, rooted liners	Camellia, azalea, and other low-fertilizer requiring crops	General production crops, privet (*Ligustrum*), crape myrtle (*Lagerstroemia*), etc.

Don Merhaut is UC Cooperative Extension Specialist for Nursery and Floriculture Crops, Department of Botany and Plant Sciences, UC Riverside.

Field observations: Boxwood blight found in Bay Area residential landscapes

by: **Steve Tjosvold**

Boxwood blight is a serious disease of most boxwood species (*Buxus* spp.), caused by the fungus *Calonectria pseudonaviculata*. Host plants also include the boxwood relatives *Pachysandra terminalis* (Japanese spurge), *Pachysandra procumbens* (Allegheny spurge) and *Sarcococca* species (sweet box); other susceptible plants may be identified in the future. First seen in the eastern United States in 2011, boxwood blight has now spread to the west coast. In California, boxwood blight has so far been identified in established boxwood hedges at three residences in San Mateo and Santa Clara counties. Nursery inspectors in California have been advised to look for symptoms of boxwood blight, but so far the disease has not been seen in California nursery stock.

Early symptoms include darkened, somewhat purplish discoloration of the foliage, and also tan to dark-brown circular leaf spots (fig. 1–2). Narrow longitudinal black lesions occur on the stems (fig. 3). Advanced disease presents as overall browning and defoliation (fig. 4).

Fig. 1. Darkened, purplish discoloration of the foliage caused by boxwood blight. *Photo*: CDFA.

The disease is primarily spread by transporting contaminated plant material (plants, decorative boxwood greenery), but spores can also be spread

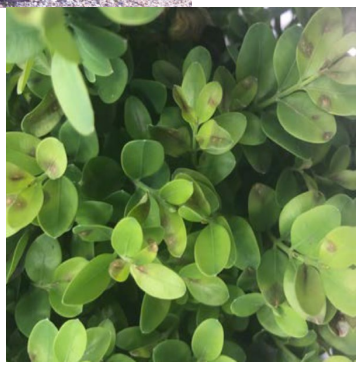

Fig. 2. Boxwood blight symptoms include tan to dark-brown circular spots on leaves. *Photo:* CDFA.

on pruning tools, clothing, equipment and anything that might have contacted infected plants. Wet or humid conditions and temperatures ranging from 64 to 80°F provide optimum conditions for infection and spread of the disease, and for defoliation of plants.

Boxwood blight is very difficult to control and eradicate. A combination of plant inspection, cultivar selection, rigorous sanitation procedures and fungicide application can be used to maintain boxwoods in a landscape where the disease is present. Some boxwood cultivars possess various levels of

Fig. 3. *Below left*: Narrow, longitudinal black lesions develop on the stems of a plant newly infected by *Calonectria pseudonaviculata* (arrow). *Photo*: CDFA.

Fig. 4. *Top right*: Boxwood blight symptomatic stems and leaves. *Photo:* CDFA.

resistance, but none are immune. The home gardener might prefer to plant boxwood alternatives such as cultivars of *Ilex*, dwarf *Picea*, *Thuja*, *Taxus, Euonymus, Myrica* and *Myrsine Africana*.

Steven A. Tjosvold
Farm Advisor, Environmental Horticulture
UC Cooperative Extension Santa Cruz County
1432 Freedom Boulevard
Watsonville, CA 95076-2796
(831)763-8013 phone, (831) 763-8006 fax
satjosvold@ucanr.edu
http://cesantacruz.ucanr.edu/

CDFA NURSERY ADVISORY REPORT

***by* Loren Oki**

The CDFA Nursery Advisory Board (NAB) met in Sacramento on March 1. The agenda, as usual, was packed but here are some highlights.

New Pests of Concern in California

- There are typically 80 detections of invasive exotic fruit flies per year in California. Last year (2016) there were 46 detections. All were transported in (since the insects cannot fly into the state) and all the detections were in metropolitan areas.
- The detection in the Ontario/San Bernardino area of *Bactrocera tau* in 2016 was the first detection of this pest in the United States. This fruit fly lays its eggs in fruit on which the larvae feed. It favors cucurbits but can be found on other plants, including bell pepper.
- Monitoring of the Japanese beetle continues in the Sacramento area.
- Eradication of the European grapevine moth was declared in August 2016.
- Asian citrus psyllid is now in West Sacramento. The huanglongbing (HLB) disease is only in Los Angeles County right now.
- Olive bark beetle (*Phloeotribus scarabaeoides*), a Mediterranean native, was detected in Riverside in 2016. This was the first detection of this pest in the Americas.
- Curtain fig psyllid (*Macrohomotoma gladiata*) was detected in Anaheim in 2016, and was the first detection of this pest in the United States.

Invasive Plants and Weeds

Santa Maria feverfew (*Partenium hysterophorus*) was found in two locations in Orange County. In addition to being a noxious weed, the American tropics native plant can cause contact dermatitis.

Disease Update

Boxwood blight caused by the fungus *Calonectria pseudonaviculata* (synonym *Cylindrocladium buxicola*) was found for the first time in California in the Palo Alto region. Information regarding this disease that was distributed at the NAB meeting can be found at https://goo.gl/EU8CYH and https://goo.gl/xB9WXs.

Quarantine Update

- Glassy-winged sharpshooter has infested all of Southern California. There is one active infestation in Santa Clara County, but eradication in that county may be declared this year. Unfortunately, there is an increased incidence of Pierce's disease in Northern California.
- Medfly quarantines continue in the San Fernando Valley (Areleta) where 3 males, 15 females and 111 larvae were found with the last detection in January.
- Asian citrus psyllid quarantines continue either partially or entirely in 29 counties. Plants grown in approved screen houses can be sold anywhere. However, plants grown outdoors can only be sold within the quarantine area.
- The HLB quarantine was expanded into San Gabriel where infected plants were destroyed. The HLB quarantine area is now 56 square miles in Los Angeles County and 26 square miles in Orange County.
- Light brown apple moth quarantines continue in San Diego, Orange, Los Angeles, Ventura, Santa Barbara and San Luis Obispo counties in Southern and Central California and in Sacramento, San Joaquin, Sonoma, Mendocino and Yolo counties in Northern California.

SANC Program

Six nurseries were approved in the first round of reviews into the SANC (Systems Approach to Nursery Certification**)** program and two additional nurseries are currently in review. There will be eight more nurseries added to the program in the second round of reviews. For information about the SANC program, see http://ucanr.edu/sites/UCNFA/files/159116.pdf and the SANC website at sanc.nationalplantboard.org/

Loren Oki is UC Cooperative Extension Landscape Horticulture Specialist, Department of Plant Sciences, UC Davis.

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Banner photo on cover (*Phytophthora ramorum* chlamydospores) by David Rizzo, Department of Plant Pathology, UC Davis.

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