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Forestry

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University**



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# Growing Healthy Seedlings

Identification and management of  
pests in northwest forest nurseries.

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# **Growing Healthy Seedlings**

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management of pests  
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Philip B. Hamm  
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Editors

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# *Abstract*

***T**his book provides an illustrated guide to the identification and management of fungi, insects, and abiotic conditions that cause problems in Northwest bareroot conifer nurseries. A key to nursery pests offers initial guidance in identification. Separate chapters address individual pests with details on recognition, damage cycle, and management practices to reduce losses. A final chapter discusses the integration of pest-management practices into the entire nursery operation. Tables of pesticides and pests controlled, a checklist of control activities keyed to the seedling growth cycle, and a glossary of terms onclude the book.*

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We have attempted to capture state-of-the-art information on nursery pest management in the Northwest. Individual chapters were written by those who know the problems best. We thank you all—your work is the heart of this book. Introductory and concluding sections were written by the editors. While we shamelessly stole the ideas of the experts, we remain responsible for any problems or disagreements. Photographic credit goes to the individual authors except where otherwise noted. File photos from Oregon State University and the USDA Forest Service were used widely.

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- Philip B. Hamm, Plant Pathologist
- Sally J. Campbell, Plant Pathologist
- Everett M. Hansen, Professor

# Introduction

## Healthy Trees!

This book focuses on the dark side of forest-nursery production. However, our emphasis on pests and problems is really just a step toward the common goal of producing healthy trees for regenerating the forests of the Northwest. Healthy trees don't grow in nurseries by accident. In fact, the same conditions that make nursery production of seedlings successful—high densities, favorable temperature and moisture, and intensive agricultural crop-management techniques—are also ideal for many of the fungi and insects that attack seedlings. Healthy trees result when pest management is integrated with the other aspects of nursery culture throughout the crop cycle. This book is about growing healthy trees by integrating nursery pest-management principles with the day-to-day operation of the enterprise.

The quality of forest-nursery stock has risen dramatically over the last 20 years. The payoff from better stock is seen in the young forests of the Northwest. With success has come higher expectations—field survival of 90 percent or better was only a dream a few years ago, and now it is the norm for most sites. A large portion of the credit goes to the managers and growers of the forest nurseries of the region. They have learned with each crop and now consistently apply those lessons. It is worth listing some of the key features of the current success, lest a new generation forget some of the practices we often take for granted.



Forest nurseries of the Pacific Northwest consistently grow high-quality seedlings that have high field survival rates on forest sites.

**1. Careful handling of seedlings.** Constant attention to lifting, storage, and handling assures that seedlings reach the field in optimal physiological condition.

**2. Better seed.** Emphasis on the collection of clean seed and the careful handling and storage of seed have improved germination vigor and greatly reduced losses from seed decay and damping-off. High-vigor seed also produces more-robust seedlings, which are better able to recover from biotic and abiotic damage.

**3. Improved nursery soils.** Better seed is sown in better ground. The worst nurseries and the worst

portions of others have been abandoned and new sites have been selected. Cultural and soil characteristics are now just as important as short-term economic considerations in selecting nursery sites—or perhaps even more. Soil management to maintain and improve the tilth and fertility of nursery soils is now everyone's priority. Pest management can be successful only where soil conditions give the seedlings at least a fighting chance.

**4. Improved pesticides and application procedures.** Most insect and nematode problems and several diseases are now effectively managed with chemicals. Growers are more aware of the need for early



detection and of the advantages of preventing problems from appearing in the first place.

**5. Attention to detail.** Nurseries are managed more professionally than ever before, and managers have a widespread understanding that the success of the whole enterprise depends on proper timing and execution of each step of the process.

Progress has been dramatic. While there is room for improvement, the emphasis of this book is on what we know rather than on what we have yet to learn.

## **Why this book? Why now?**

There are other publications on nursery pest management. These are helpful to many managers and growers. References to other sources of information follow each chapter. Our book focuses on problems of bareroot nurseries of the Pacific Northwest. The tree species, the climate, and the cultural practices of this region are different from those in other tree-growing areas, even British Columbia. Consequently, the diseases and insect pests and the strategies to manage them are different. We hope that the regional focus of this work allows it to be specific in its identifications and recommendations.

And why now? In recent years there has been an unprecedented effort from universities, the forest services of the United States and Canada, the states of Oregon, Washington, Idaho, and California, and the nurseries themselves to understand the causes of nursery problems and ways to reduce their effects. This effort has been brought on by the ever-increasing value of seed and seedlings, and by the need for the appropriate seedlings to be available on time for reforestation of cut-over sites. The information is scattered, and in some cases is not even written down. Now is the time to summarize what we have learned and make it generally available. And

there is nothing like writing a book to highlight the remaining gaps in our understanding. This book, therefore, is also the springboard for future improvements in pest-management research and development.

## **Where to find more help**

In many cases you will need more information to apply or confirm what you learn here. Any of the following sources will be helpful in providing more, or updated, information:

- Philip Hamm, Everett Hansen, or the Plant Clinic  
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# Key to Seedling Damage

## INTRODUCTION TO KEY

This key was prepared to help in the identification of some of the common causes of seedling damage in bareroot nurseries. Most of the damaging agents identified in this key are described in greater detail in the chapters that follow.

Before using the key, carefully observe the seedlings in place to note patterns of damage in the field, in the seedbed, and on the individual seedlings. Then gently dig and examine a sample of damaged seedlings. Dig a sample of healthy seedlings as well for comparison.

Remove soil from the roots by gently shaking or washing them. Examine stem and root tissues for obvious signs of insect feeding or mechanical damage (pieces of bark missing, entire roots missing) or abnormalities (shrunken areas, swellings, galls). Then examine for internal discoloration (indicating tissue death) by slicing downward from the top of the shoot to the root collar with a sharp knife, exposing the cambium and xylem. The roots should also be scraped or sliced longitudinally to expose the stele. Examine needles with a hand lens if necessary for fungal structures (mycelium or fruiting bodies) or for signs of insects (webbing or insect larvae or adults).

The key outlines symptoms in a progression from the general to the specific. The general symptoms are identified with a system of numbers and letters that guides the reader through the key in a narrowing search. Specific symptoms are identified with the likely cause.

To use the key, begin with number 1. Find the statement that best fits the observed symptoms. Then proceed to the section that corresponds with the number directly across from this statement. Repeat this procedure until the likely cause is identified.

*For example, number 1 offers three possible alternatives:*

- 1a. damage seen in first growing season
- 1b. damage seen in second or third growing season, and
- 1c. damage seen after storage.

If the damage at your nursery is seen in the second or third growing season (the second alternative), you would proceed to number 20 of the key. Number 20 also offers three alternatives:

- 20a. entire above-ground portion of seedling dead,
- 20b. only part of seedling top is dead or damaged, and
- 20c. none of seedling is dead but parts are discolored, deformed, or stunted.

If your seedlings are deformed but not dead, you would proceed to number 35. Here again you would choose the one statement of three that best fits your situation. If the needles on your seedlings are twisted, go back to number 14. Depending on the most specific symptoms listed here, your problem is likely either lygus bugs or pesticide damage.

|  |           |
|--|-----------|
| <b>1a. Damage seen in first growing season .....</b>           | <b>2</b>  |
| <b>1b. Damage seen in second or third growing season .....</b> | <b>20</b> |
| <b>1c. Damage seen after storage .....</b>                     | <b>37</b> |

## First growing season

|  |    |
|--|----|
| 2a. Entire above-ground portion of seedling dead or did not emerge .....   | 3  |
| 2b. Only part of seedling dead (branches, needles, or upper stem) .....    | 11 |
| 2c. None of seedling dead but parts discolored, deformed, or stunted ..... | 13 |

### Entire top dead or poor emergence

|   |                                       |
|---|---------------------------------------|
| 3a. Seed germinates poorly. Low density after emergence .....   | 4                                     |
| 4a. Ungerminated seed rotten .....  | Seed fungi                            |
| 4b. Non-emerged germinants rotten .....   | Damping-off                           |
| 4c. Germinants dead. Presence of 3-7 mm yellow-white<br>maggot near germinant. Seen when soils<br>cold and wet .....  | Seedcorn maggot.<br>See Minor Insects |
| 3b. Seedlings fallen over and lying on ground .....   | 5                                     |
| 5a. Seedling tissue collapsed near soil line.<br>Roots still in soil. Mortality occurs shortly<br>after emergence .....   | 6                                     |
| 6a. Killed tissue is water-soaked and constricted .....   | Damping-off                           |
| 6b. Killed tissue appears as white spot or streak<br>on stem. Damage seen after clear weather with<br>high soil surface temperatures (>49°C, 120°F) ...   | Heat injury                           |
| 5b. Seedling roots wholly or partially out of soil.<br>No dead tissue near soil line. Occurs after<br>soil surface has repeatedly frozen and thawed .....   | Frost heaving.<br>See Cold Injury     |
| 3c. Seedling is dead but does not fall over.<br>Mortality is seen after tissue lignifies, usually<br>mid-summer through early fall .....  | 7                                     |
| 7a. Roots healthy .....   | 8                                     |
| 8a. Stem girdled by bark removal. No necrosis<br>associated with wound; some callusing may<br>be found around wound. Pattern of damage<br>may be related to various cultural<br>operations .....  | Mechanical damage                     |
| 8b. No bark removed on stem. Necrotic area on<br>stem at soil line (or slightly above or below).<br>Stem shrunk where necrotic. Primarily on<br>Douglas-fir; also on other species .....  | Fusarium<br>hypocotyl rot             |
| 7b. Roots decayed or diseased .....   | 9                                     |
| 9a. Roots decayed from tips upward. Laterals<br>lacking. Existing roots dark and swollen.<br>Douglas-fir and pines most commonly<br>affected .....  | Fusarium root rot                     |
| 9b. Root tips, laterals, and root crown necrotic<br>and blackened. Microsclerotia visible with<br>hand lens in dead inner bark of roots and<br>lower stem. Affects sugar pine, Douglas-fir,<br>giant sequoia, and true fir primarily.<br>Mostly in California nurseries ..... | Charcoal root disease                 |

- 7c. Roots missing. Main stem may be cut off just below ground line ..... 10
- 10a. Damage seen shortly after emergence while seedling tissue still succulent. Seedling stem cut off at or just below soil line ..... Cutworms
- 10b. Damage seen in late spring and summer. Damage variable; roots not all severed at the same point and not all seedlings affected in one area. White grubs may be found in soil around roots. .... June beetle
- 10c. Damage seen following root undercutting or wrenching. Roots all severed at same point. All seedlings in one area or one part of bed affected ..... Mechanical damage

### **Part of top dead**

- 11a. Seen early in first growing season while seedling still succulent. Needles chewed or missing ..... Cutworms
- 11b. Seen in middle to late part of first growing season ..... 12
  - 12a. Cankers on stem or branches. No mycelium on killed tissue. Portion above canker wilted or dead. Seen in late summer or fall on Douglas-fir ..... Upper stem canker
  - 12b. Cankers on stem rare. Needles and small branches killed. Gray mycelium on killed tissue. Most frequent in lower crown of densely sown seedlings or on tissue that was previously killed by other agents (e.g., frost) ..... Gray mold
  - 12c. No cankers on stem. More-exposed or succulent needles and stem tissue killed. Seen several days to several weeks after frost. Often distinct pattern of damage in field ..... Frost damage.  
See Cold Injury

### **Deformity, discoloration, stunting**

- 13a. Stunted seedlings; needles short and green; premature budset ..... Phosphorus deficiency.  
See Mineral Nutrient Problems
- 13b. New needle growth deformed; needles twisted ..... 14
  - 14a. Twisted needles also thickened. Terminal bud often killed, resulting in multiple leaders the following year. Vertical scars on new stem tissue. Affects several species including Douglas-fir and pines ..... Lygus bugs
  - 14b. Twisted growth associated with pesticide applications, particularly herbicides with hormone-type action. Damage occurs at same time on all seedlings; usually some pattern of damage in field or seedbed ..... Pesticide phytotoxicity
- 13c. Needles discolored or scorched-appearing but not dead or deformed ..... 15
- 15a. Needles yellow; seedlings may be stunted ..... 16
  - 16a. Roots not fully developed or not healthy ..... 17

|  |   |
|--|---|
| 17a. Roots decayed or missing .....  | 18  |
| 18a. Random distribution of symptomatic<br>seedlings .....   | 9 (for 1+0),<br>25 (for 2+0)                                |
| 18b. Circular or irregular patches of symptomatic<br>seedlings .....   | 19  |
| 19a. Extreme proliferation of feeder root<br>laterals (witches' broom) above feeder<br>root terminals. Eventually all feeder roots<br>destroyed. Douglas-fir and true firs<br>most commonly affected | Root lesion nematode.<br>See Nematodes                      |
| 19b. Roots dark, swollen, and often club-tipped.<br>Few laterals. Spruce, western hemlock,<br>noble fir, and Douglas-fir affected .....  | Baker dagger<br>nematode.<br>See Nematodes                  |
| 17b. Roots poorly developed. Dead roots, if present, are<br>gray, blue, or black internally. Associated<br>with compacted or waterlogged soil .....  | Compacted or<br>anaerobic soil.<br>See Soil Compaction      |
| 17c. Roots not fibrous. Needle tips may be<br>scorched. Associated with high levels<br>of soluble salts in surface soil or irrigation water .....  | Salt injury   |
| 16b. Roots healthy; stem girdled .....   | 8 (for 1+0),<br>24 (for 2+0)                                |
| 16c. Roots and stem healthy. Yellowing associated with<br>undecomposed organic material or poor nutrient<br>status of soil or foliage  | Nitrogen deficiency.<br>See Mineral Nutrient Problems       |
| 15b. Needles discolored other than yellow.<br>Nutritional analysis of seedling tissue indicates<br>nutrient imbalance, deficiency, or toxicity   | Various nutrient problems.<br>See Mineral Nutrient Problems |

## Second or Third Growing Season

|   |    |
|---|----|
| 20a. Entire above-ground portion of seedling dead .....                           | 21 |
| 20b. Only part of seedling top is dead or damaged .....                           | 27 |
| 20c. None of seedling is dead but parts discolored,<br>deformed, or stunted ..... | 35 |

## Entire Top Dead

|  |                   |
|--|-------------------|
| 21a. Seedling stem girdled at or below soil line.<br>Roots healthy .....   | 22                |
| 22a. Death of seedling seen before or just<br>after bud break in spring .....  | 23                |
| 23a. Necrotic area on stem just below soil line.<br>Associated with soil collars and low, wet<br>areas in the nursery. Douglas-fir and true<br>firs affected ..... | Lower stem canker |

- 23b. Stem and foliage desiccated; no clear margin between healthy and necrotic tissue in cambium and xylem. Affected seedlings exposed to dry, cold conditions previous winter ..... Winter desiccation, freeze damage. See Cold Injury
- 23c. Dormant buds killed. Dieback progresses down stem from buds. Associated with heavy soil splash during winter. True firs affected ..... Phoma blight
- 22b. Death or yellowing of seedling seen in late fall or winter. Areas of bark and wood removed above or below soil line ..... 24
- 24a. Bark and wood of stem chewed, giving a ragged appearance. Damage seen 24 mm (1 inch) above and below soil surface with some upper roots chewed. Damage seen in patches in bed; may not be noticed until lifting. Douglas-fir, true firs, spruce affected ..... Cranberry girdler. See Sod Webworm
- 24b. Stem girdled by bark removal. No necrosis associated with wound; some callusing may be found around wound. Pattern of damage may be related to various cultural operations ..... Mechanical damage
- 21b. Roots decayed, missing, or girdled ..... 25
- 25a. Roots missing. Main stem may be cut off just below ground line. Damage seen in late spring and summer. Damage variable; roots not all severed at the same point and not all seedlings affected in one area. White grubs may be found in soil around roots ..... June beetle
- 25b. Roots missing. Damage seen following root undercutting or wrenching. Roots all severed at same point. All seedlings in one area or one part of bed affected ..... Mechanical damage
- 25c. Bark removed from roots, often girdling them. Damage seen late summer through lifting ..... Root weevils
- 25d. Roots decayed ..... 26
- 26a. Some or all of roots dead; cambium on dead roots discolored reddish-brown. Patches of stunted dead or yellowed seedlings seen in low, poorly drained areas. Mortality occurs throughout growing season. Most species, especially Douglas-fir and true firs affected ..... Phytophthora root rot
- 26b. Lateral root(s) killed, with necrosis frequently extending to taproot. Seen at bed ends and in wet areas. Symptoms appears in spring. Only Douglas-fir affected ..... Fusarium root necrosis
- 26c. Root tips, laterals, and root crown necrotic and blackened. Microsclerotia visible with hand lens in dead inner bark of roots and lower stem. Affects sugar pine, Douglas-fir, giant sequoia, and true fir primarily. Mostly in California nurseries ..... Charcoal root disease

## Part of Top Dead

- 27a. Only needles damaged ..... 28
- 28a. Needles killed ..... 29
- 29a. Mainly lower needles (senescent or previously killed) affected. Gray mycelium seen on killed tissue ..... Gray mold
- 29b. On larch only. Lower needles killed. Reddish-brown discoloration of needle tips or entire needle. Fungus growth cannot easily be seen on needles. .... Larch needle cast
- 29c. New flush killed. Follows below-freezing temperatures ..... Frost damage. See Cold Injury
- 29d. Needles wholly or partly killed. A pattern of damage can be seen in nursery beds. Damage seen shortly after pesticides applied. All species affected but white pines especially sensitive ..... Pesticide phytotoxicity
- 28b. Needles not killed. Discolored spots on needles ..... 30
- 30a. Yellow spots on upper surface of needle; rust-red clusters of spores on lower surface. Affected seedlings in close proximity to *Populus* trees. Only Douglas-fir affected ..... Melampsora needle rust. See Needle Rusts
- 30b. Light green to brown spots or bands on needles in late summer or fall. Needles turn brown and drop the following spring. Only pines affected ..... Lophodermium needle cast
- 28c. Insect feeding or presence of insect on foliage ..... 31
- 31a. Needles missing or showing evidence of insect feeding ..... 32
- 32a. Feeding on new growth only ..... Root weevils
- 32b. Feeding on old and new growth. Presence of grasshoppers ..... Grasshoppers. See Minor Insects
- 31b. Insect webbing on new needles. Small green larvae in or near webbing ..... Leafrollers. See Minor Insects
- 31c. White, cottony tufts seen on foliage or bark ..... 33
- 33a. On Douglas-fir; white, waxy-covered aphid found on needles ..... Cooley spruce gall adelgid. See Minor Insects
- 33b. On pines; white, waxy-covered aphid found on bark of stem and branches and on foliage ..... Pine bark adelgid. See Minor Insects
- 27b. Branches or upper portion of stem damaged or killed ..... 34
- 34a. Only Douglas-fir affected. New growth killed. Stem canker found at junction between 1-year-old and 2-year-old growth ..... Phomopsis canker
- 34b. Only pines affected. Tips of shoots killed during spring and summer of second year. Killed shoots frequently crooked over. Black fruiting bodies may be seen on killed needle or stem tissue ..... Sirococcus tip blight or Phoma tip blight

---

## Deformity, Discoloration, Stunting

- 35a. New needle growth deformed; needles twisted ..... 14
- 35b. Needles discolored or scorched-appearing but  
not dead or deformed ..... 15
- 35c. Stem swollen but no cankers, lesions, or necrosis  
near swollen area ..... 36
- 36a. Globose to pear-shaped swellings on stem or  
branch. Seen late in second growing season.  
On ponderosa, lodgepole, and other two- and three-  
needle pines ..... Western gall rust
- 36b. Swellings on stem, frequently near soil line.  
Symptoms appear on all seedlings at same time.  
Associated with pesticide applications ..... Pesticide phytotoxicity

## After Lifting or Storage

- 37a. Fungus growth seen on stem, roots, or foliage  
of seedlings ..... 38
- 38a. Tissue associated with fungus growth  
water-soaked, yellowing, or dead. Seen after  
seedlings have been in lifting tubs or in  
storage for a period of time ..... Storage molds
- 38b. Tissue associated with fungus growth healthy ..... 39
- 39a. Light brown to black leathery fungus  
fruiting bodies encircling the lower stem  
of the seedling ..... *Thelephora terrestris*  
(a mycorrhizal fungus)
- 39b. Fungus mycelium on roots; various  
colors and textures. Root tips associated with  
mycelium are short, smooth, and lobed; they  
are often golden brown in color ..... Mycorrhizae
- 37b. No fungus growth seen on seedling, but parts  
of stem, root system, or foliage may be water-  
soaked, yellowing, or dead. Seen after  
seedlings have been in lifting containers or  
in storage for a period of time ..... Adverse storage conditions  
or storage molds
-



## CHAPTER ONE

# Damping-off

**Kenelm Russell**

Damping-off is the disease term used for fungal-caused mortality during those first critical few weeks from germination to just after seedling emergence. The soil-inhabiting fungi associated with damping-off are capable of causing rapid decay and mortality of seeds and germlings. These fungi are not host-specific.

## Disease and hosts

Many species of fungi, often common soil saprophytes, are associated with damping-off and root rot. They become pathogenic when temperature, moisture, soil pH, and other conditions become favorable. In Pacific Northwest forest nurseries, *Pythium* and *Fusarium* species are the most common damping-off fungi. Others are *Rhizoctonia solani*, *Macrophomina phaseoli*, *Botrytis cinerea*, and *Phoma*, *Alternaria*, and *Phytophthora* species. In general, *Pythium* species cause problems early in the season when soils are cool and wet, while *Fusarium* species cause problems later when soils are warmer and moist to semi-dry. Exceptions occur, however, and any of these fungi can and do cause disease at any time during the growing season.

Damping-off fungi occur naturally in nearly all crop and forest soils. They are found worldwide in temperate and tropical zones alike. No conifer or hardwood is known to be resistant to damping-off in the Pacific Northwest. Those species or seedlots that germinate quickly and grow fast may sustain less damage from damping-off than slow-emerg-



**Figure 1-1.** Postemergence damping-off of Douglas-fir germlings. Fungal invasion of succulent stem tissue causes collapse of infected tissue, so that seedlings fall over.

ing, slow-growing species. Still, it is safe to assume that all nursery-grown tree species are susceptible to damping-off fungi.

## Symptoms

Damping-off is defined as the fungal invasion of the succulent tissue of germinants or seedlings that leads to decay and early death. Damping-off attacks seedlings both before emergence (preemergence damping-off) and after (postemergence damping-off) and, depending on conditions, usually occurs within 30 to 45 days after sowing.

The only evidence of preemergence damping-off in nursery beds is

that the germinating seedlings are sparse and patchy. This phase is difficult to detect, but may sometimes be diagnosed by digging up seeds that have not emerged and checking to see whether seeds or germinants are decayed or withered.

Postemergence damping-off, which occurs in the cotyledon stage, causes seedlings to wither and collapse (Figure 1-1). When the succulent root-collar tissue or the roots are penetrated by the pathogen, the disease is referred to as soil-infection damping-off. When the fungal invasion occurs higher on the stem or cotyledons, it is called top-infection damping-off.

The most obvious indicator of postemergence damping-off is the

collapse of the seedling. It may be possible to tentatively identify, at least to genus, the fungus responsible. Stem tissues of seedlings infected by *Pythium* sometimes separate around the root collar, with the epidermis sloughing away from the inner xylem tissue as an open shirt collar falls away from the neck of the wearer. The trees then fall over. Seedlings infected by *Fusar-*

**Damping-off symptoms appear:  
1 +0  
Late spring through early summer**

*ium* undergo a softening of the root-collar tissue, and the trees fall over at the point of softening without separation of the stem tissues. Seedlings with suspected *Fusarium* infection may be incubated overnight at room temperature in a moistened paper bag to produce a bloom of sickle-shaped macroco-

nidia that can be easily identified under a microscope (see Figure 2-3).

In broadcast-sown beds, which are uncommon in Pacific Northwest nurseries, seedlings may die in irregular bull's-eye patches, with the centers containing mostly fallen trees and the borders containing trees with early symptoms. In drill-seeded beds, the mortality pattern usually runs along the rows for a distance, then abruptly stops. Adjoining rows may be affected, showing a patchy effect (Figure 1-2).

It is not unusual for forest pathologists to isolate one or more species of damping-off fungi from apparently healthy seedlings within the first few weeks of germination. The fungi may be found either on root surfaces or within internal tissues and may have come from infested seed or surrounding soil. These symptomless seedlings usually remain healthy as long as moisture stress is low and other growing conditions are optimum.

## Fungus biology

Damping-off fungi are inhabitants of the soil. They can be spread by movement of soil on equipment or seedlings, by cultivation, or by water. Infection occurs when seedling roots grow next to fungal inoculum, such as chlamydospores, sclerotia, or oospores. These structures then germinate and hyphae invade the seedling cells. Fungal invasion causes collapse and disintegration of cells and death of the seedling. The fungus may continue to develop in and utilize the killed tissue, often producing secondary inoculum, such as conidia, on the surface of the dead seedling. Mycelium, spores, or

**Damping-off may be confused with:  
Cutworm damage  
Frost heaving  
Fusarium hypocotyl rot  
Heat damage  
Seedcorn maggot damage**

other structures survive and overwinter in seedling tissue or other organic material in the soil. Viability of overwintering inoculum is dependent on a number of factors, including soil moisture and temperature.

## Loss potential

Damping-off fungi can cause significant losses in forest nurseries. Losses may be large one year and minor the next. Mortality from preemergence damping-off can be estimated by calculating the difference between the number of seedlings and the number of seeds sown, after other factors, such as percent germination or bird depredation, are accounted for. Postemergence damping-off is best determined by marking small plots and counting mortality every few days. Individual



**Figure 1-2. Patchy, uneven density is the result of significant levels of pre- and postemergence damping-off.**

Yes can be marked with toothpicks monitor whether the disease is increasing, subsiding, or responding to treatment. Losses from preemergence damping-off often range from 15 to 40 percent of sown seed, while postemergence losses may be an additional 10 to 20 percent. Growers typically oversow to ensure a satisfactory crop.

Damage may be heavy in seedling beds that previously contained transplants or other agricultural crops. In fact, new nursery sites developed from cleared forest soils tend to have fewer damping-off problems than those established on previous agricultural croplands.

In addition to the direct losses of bed stock, indirect losses may be reckoned in shortages of healthy seedlings for outplanting on forest sites.

## Management

The best defense against damping-off fungi is an effective and conscientious disease-prevention program. This includes knowledge of soilborne disease populations, a well-orchestrated pesticide program, and careful attention to environmental conditions in the nursery. The nursery environment is the strongest influence on the proliferation of damping-off fungi. Soil moisture, timing and amount of irrigation, air and soil temperatures, method and timing of sowing, depth of soil over seed, soil pH, combinations of soil fungi and nematodes, timing and type of nutrients applied, type of organic matter, type of cover crop, history and pattern of pesticide use, and many other factors affect the incidence and severity of damping-off.

It is to the grower's advantage to bring the entire crop through its initial growth stages rapidly and evenly in order to narrow the damping-off infection "window." No single factor alone governs control of the disease, but good management will take the following factors into account:

### SOIL MOISTURE AND DRAINAGE

Ideally, nurseries should be located on light, well-drained soils. Wet soil generally favors damping-off. Depth of irrigation is critical to young seedlings, especially during hot weather. It is important to irrigate deeply enough for water to reach seedling roots—a depth that increases steadily as the roots grow—but not so much that the soil is saturated. Too-shallow watering stresses tender roots in moisture-deficient lower soil layers, while creating a warm, wet upper soil layer that favors the buildup of damping-off fungi. Soil moisture and rooting depth should be monitored regularly during the growing season. Cool weather that prolongs germination, or hot weather that speeds it up, require particular attention to watering.

### TIMING OF SOWINGS

Sowing when temperatures are warm enough to promote rapid, even germination tends to reduce problems with damping-off. Warm-weather sowing requires constant diligence in controlling irrigation.

### SOIL pH

Damping-off fungi thrive in neutral to alkaline soils. A soil pH of between 5.2 and 5.7 (moderately acid) not only helps prevent damping-off problems but is ideal for growing Pacific Northwest conifer species. Aluminum sulfate drenches, sulfur (200 to 500 pounds/acre), and acid peat applications can be used to maintain the acid condition of the soil. When aluminum sulfate is used, beds should be kept moist to prevent burning of the roots.

Irrigation water that is even slightly alkaline can, over a period of years, decrease soil acidity. The change usually occurs slowly because of the tremendous buffering ability of the soil. It can be reversed by acidifying the water with either sulfuric or phosphoric acid. The acidification process can be speeded up by adding sulfur to the soil and

then maintaining the pH with acidified water.

### SOIL MICROFLORA

No two nurseries are alike in their makeup of soil organisms. Each has its own combination of soil microflora, consisting of bacteria, fungi, nematodes, and insects, and each combination influences the population of pathogenic fungi in the soil, the amount of infections, and the expression of disease symptoms. Growers should learn the soil microflora "personality" of their nurseries.

### NUTRITION

Nitrogen applications made too early promote damping-off. Germinating seed and new germlings do not need much supplemental nutrition; the endosperm contains sufficient food to get seedlings well on their way.

### MULCHES AND COVER CROPS

Cover crops grown and turned under just prior to sowing conifers may, depending on their type, retard or encourage damping-off problems. Legume cover crops promote large populations of damping-off fungi, grass crops somewhat smaller populations. Bare fallowing discourages the buildup of potential pathogenic fungi in the soil.

### ASSAYS FOR SOILBORNE DISEASES

Assays for soilborne pathogens measure populations of particular fungi in the soil. Soil assays have been developed for *Pythium*, *Fusarium*, *Macrophomina*, and *Phytophthora* species (see the passage on monitoring of fungi in Chapter 33, Principles of Integrated Pest Management). Although population levels of these fungi indicate potential risk and the severity of disease in future crops, they are not reliable predictors of crop loss. The grower should use the assay as a warning signal to give an indication of potential problems and to help determine disease prevention measures, such as fumigation.

### SOIL FUMIGATION

Fumigating soil prior to sowing is a common practice in Pacific Northwest nurseries. Several different materials have been used successfully, including dazomet, methyl isothiocyanate/1,3-dichloropropene, and mixtures of methyl bromide and chloropicrin. Fumigation decreases *Fusarium* and *Pythium* populations sometimes to near zero. Methyl bromide with 33 percent chloropicrin will hold these pathogens in check for most of the first growing season. Follow-up disease control is done as needed with a carefully prescribed fungicide application plan.

### FUNGICIDES

Treatment of seed with fungicides is not recommended. In previous years seed treatment was customary, but fungicides applied to the seed coat offer little or no protection to the emerging seedling. In addition, some seed treatments are phytotoxic.

Although the effectiveness of fungicides to control damping-off is highly variable (see the passage on fungicides in Chapter 33), many growers use them. Several fungicides are registered for use in forest nurseries to control soilborne diseases. Certain fungicides or combinations of fungicides seem to work better in one nursery than another. The fungicide metalaxyl has systemic properties and may be used prior to sowing to reduce populations of *Pythium* and *Phytophthora* in the soil. Metalaxyl is available in granular and liquid formulations.

The first post-plant fungicide application should be made when most seedlings have emerged and the seeds begin to drop from cotyledon leaves. A good all-purpose preventive treatment for damping-off is a 50-50 mixture of captan and benomyl applied as a drench at rates recommended on the label. If frequent applications are planned, alternation of the captan-benomyl mix with other fungicides is advised

to minimize the buildup of resistant pathogens.

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## CHAPTER TWO

# Fusarium Hypocotyl Rot

**Philip B. Hamm**

## Disease and hosts

Fusarium hypocotyl rot is caused by the fungus *Fusarium oxysporum*. It occurs primarily on Douglas-fir, and to a lesser extent on Shasta red fir, western larch, sugar pine, white fir, and ponderosa pine, in the Pacific Northwest. Symptoms usually begin to appear after the first period of high temperatures (above 32 degrees C or 90 degrees F) in late June or July. The disease continues to cause damage through August or September. Fusarium hypocotyl rot is the greatest single cause of loss of postemergent seedlings in Pacific Northwest nurseries.

**Fusarium hypocotyl rot may be confused with:**

**Damping-off**

**Fusarium root rot**

**Heat damage**

**Mechanical damage**

## Symptoms

Random mortality begins in seedbeds in June (Figure 2-1) and continues through October. Mortality is highest in July and early August. Top symptoms are similar to those caused by Fusarium root rot, but careful observations made just as the seedling tops begin to turn yellow reveal a discolored region of dead tissue on the hypocotyl, that portion of the stem between the ground and the two



**Figure 2-1. Random mortality of Douglas-fir in the field due to *Fusarium hypocotyl rot*. Photo courtesy of Alan Kanaskie.**

lowest needles, or cotyledons (Figure 2-2). This discolored region quickly expands, girdling the stem and killing the top while the roots remain healthy. The roots then become discolored and decay in the same way as do roots infected with Fusarium root rot. Seedlings remain erect. White mycelia with pink or orange spore pustules can be seen occasionally on infected seedlings at or above the soil line. The sickle-shaped spores are readily identifiable under a microscope (Figure 2-3).

## Fungus biology

*Fusarium oxysporum* is a common soilborne fungus. It forms thick-walled resting structures called chlamydospores in plant material

such as residue from previous crops or weeds. Chlamydospores are known to germinate in the presence of susceptible tissue such as roots, and presumably act as one source of primary inoculum for infection. Another potential source of infection is conidia, which are thin-walled, sickle-shaped spores that can be produced in large numbers on infected tissue. It is not known whether conidia serve as primary or secondary inoculum. Infection occurs shortly after the seeds germinate, but symptoms usually do not develop until mid-summer.

Infection by *F. oxysporum* does not necessarily lead to mortality. Recent findings indicate that most healthy-looking seedlings have *Fusarium* already on or in the hypocotyl region at emergence or soon after. If

seedlings are not stressed by high soil temperatures or low moisture, they remain healthy.

## Loss potential

Greater than 50 percent mortality has been reported in sugar pine, red fir, and white fir in two California nurseries. Damage on Douglas-fir is highly variable. During 1983 and 1984, losses at three nurseries in western Oregon and Washington averaged nearly 10 percent, whereas in 1987, five of seven nurseries surveyed reported less than 3 percent loss and the other two reported 10 percent loss. In the past, losses due to *Fusarium* hypocotyl rot were probably attributed to Fusarium root rot because the above-ground symptoms are similar, and the causal agent is the same.

**Fusarium hypocotyl rot  
symptoms appear:**

**1 + 0**

**Summer through early fall**

## Management

Soil fumigation in the fall prior to sowing is likely the best consistent control of this disease when coupled with proper irrigation practices. In nurseries where comparisons were made between seedling survival in non-fumigated and fumigated soil during the first growing season, higher survival was most often found in fumigated areas. While mortality in those cases included all losses, a component was *Fusarium* hypocotyl rot. The kind of soil fumigant used probably does not matter.

A carefully timed watering schedule is also important. Warm soil temperatures seem to increase mortality because seedling moisture stress is also increased. Deep, infrequent watering, enough to ensure complete wetting of the root



**Figure 2-2. Healthy (left) and *Fusarium*-infected Douglas-fir seedlings. Note that the diseased seedling on the right has a white taproot but a brown and sunken stem below the cotyledons. Photo courtesy of Alan Kanaskie.**

zone, reportedly lowers disease losses by decreasing moisture stress and heat stress on seedlings. In one study, seedlings that were watered more heavily, because they were closer to sprinkler heads, had less disease. Mulching may help by reducing loss of moisture from the soil.

Several fungicides have been tested for control of this disease, but with limited success—probably because there is no good systemic fungicide, and none that can adequately penetrate the soil to protect roots or hypocotyl before emergence. A fungicide with these characteristics is needed, particularly to fight infection occurring before or at seedling emergence. Benomyl, applied to seedbeds at 2-week intervals beginning after emergence and continuing through September, has been most effective. The efficacy of late-season applications of benomyl has not been shown.

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**Figure 2-3. Microscopic spores produced by *Fusarium oxysporum*.**

## CHAPTER THREE

# Fusarium Root Rot

Jack R. Sutherland

## Disease and hosts

*Fusarium* root rot of bareroot nursery seedlings is most often attributed to the fungus *Fusarium oxysporum*, although other *Fusaria* have sometimes been implicated in root rots of conifer nursery seedlings. All species of conifers grown in Northwest nurseries except western redcedar are susceptible to *Fusarium* root rot. Douglas-fir and pines are particularly vulnerable. In general, *Fusarium* root rot losses are confined to the first year of seedling growth. Transplants are usually not killed, but growth may be stunted.

**Fusarium root rot may be confused with:**

**Baker dagger nematode damage**

**Charcoal root disease**

**Fusarium hypocotyl rot**

**June beetle damage**

**Root-lesion nematode damage**

## Symptoms

In seedbeds, *Fusarium* root rot first becomes evident in early to late August; losses can continue through late autumn. Transplants can exhibit disease symptoms during the first flush of growth. Disease symptoms are generally the same in all hosts. The disease is normally confined to individual, randomly distributed seedlings, but sometimes small patches of six to eight seedlings are affected.



**Figure 3-1.** A 1+0 Douglas-fir killed by *Fusarium* root rot. The shoot often assumes a crozier shape.

Shoot symptoms are usually the first to be noticed. Shoot growth becomes stunted and yellowed, and the shoot terminal often assumes a crozier shape. The affected shoots dry out and their needles become reddish-brown (Figure 3-1).

Root systems of diseased seedlings lack laterals; roots that are present are dark and swollen. The rotted cortex can be pulled off to reveal the dark, killed cambium (Figure 3-2). The rot may extend partway up the seedling hypocotyl, but is not confined to the hypocotyl as with *Fusarium* hypocotyl rot.

*F. oxysporum* produces a profusion of both macro- and micro-conidiospores both in and on diseased tissues. The former are multiseptate and slightly sickle-shaped and the latter are single-celled and oval to kidney-shaped. Sometimes sporodochia are evident in splits or cracks in diseased stems slightly above the ground line.



**Figure 3-2.** *Fusarium* root rot of ponderosa pine. Notice the dark cortex, which when removed reveals a discolored cambium. The fungus has progressed upward on the root to near the soil line.

## Fungus biology

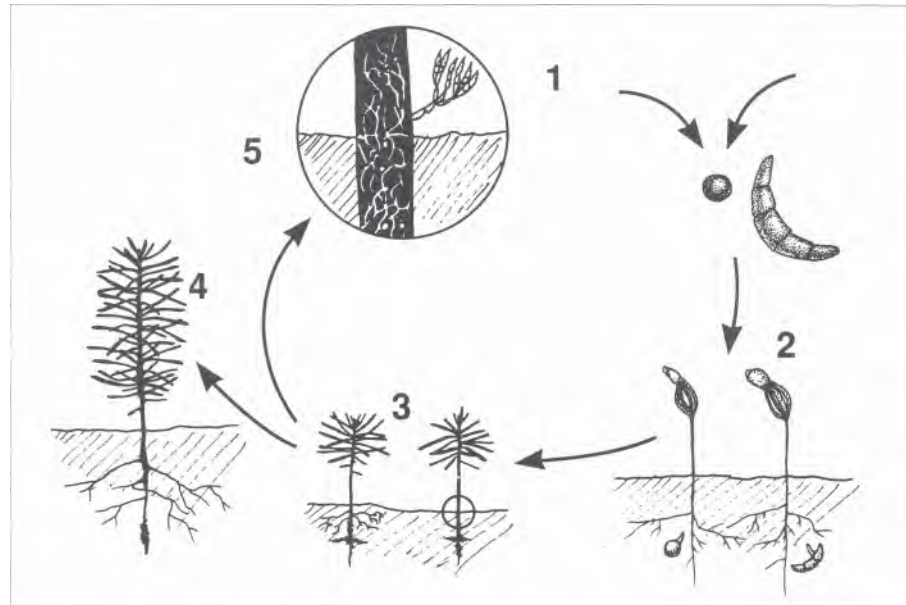
Round, thick-walled, single-celled chlamydospores are produced in abundance in killed tissues or pieces of colonized organic matter in the soil (Figure 3-3). Chlamydospores allow the pathogen to lie inactive in the soil when it lacks a suitable host and thus to survive periods of unfavorable conditions such as drought and low temperatures.

The pathogen becomes active when a seedling root grows near chlamydospores. Infection is

thought to occur early in the growing season, with the pathogen remaining inactive within the roots of 1+0 seedlings until later in the season (August), when it ramifies throughout the roots. In transplants, the pathogen can be active much earlier in the season (June).

Warm weather favors the pathogen; indeed, losses are greatest in years with hot summers. High temperatures stress the host, thereby increasing its susceptibility. Losses are greater among seedlings grown at higher densities, which indicates that host stress plays a role in the development of *Fusarium* root rot.

**Fusarium root rot  
symptoms appear:  
1+0, rarely 2+0  
Transplants  
Summer through early fall**



**Figure 3-3.** The disease cycle of *Fusarium* root rot. (1) Chlamydospores released from decaying roots, or macroconidia formed on dead stems and transported by wind or in soil, germinate (2) when contacted by seedling roots. (3) Infected roots may be killed. Some seedlings survive by producing new roots above the infection (4). The fungus forms chlamydospores in dead roots and may sporulate on killed stems (5).

materials that harbor chlamydospores. Reducing soil temperature and seedling moisture stress by shading and frequent irrigation (in well-drained soils) may help reduce the severity of *Fusarium* root rot.

Treatment of seeds with fungicides is usually not worthwhile because the fungicide loses its effectiveness before infection occurs. It does little good to apply fungicide drenches when symptoms appear because by that time the fungus is already in the root system. However, soil fumigation with such chemicals as methyl bromide plus chloropicrin is effective against *Fusarium* root rot. Fumigation is recommended only for those nurseries in which the disease is a component of an overall pest problem that includes weeds, insects, and other diseases.

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## Loss potential

The disease causes losses in most nurseries throughout the Pacific Northwest. *Fusarium* root rot can affect 10 to 30 percent of the crop, and sometimes more. The disease not only kills seedlings, but reduces the growth and vigor of those that are infected but survive with only part of their roots rotted. Survival rate of such seedlings is often poor after they are outplanted.

## Management

*Fusarium* root rot is a difficult disease to manage. There is no satisfactory way to predict outbreaks. However, certain cultural practices offer some protection from the disease. These include identifying high-risk areas and using them for less susceptible species, minimizing the carryover of inoculum-laden root pieces between crops, and reducing or eliminating organic



## CHAPTER FOUR

# Gray Mold

**Kenelm Russell**

Gray mold, caused by *Botrytis cinerea*, is a disease that causes blight of flowers, leaves, and shoots, and decay of fruit in hundreds of woody and herbaceous plants. It occurs worldwide, flourishing where the air is moist and stagnant.

Gray mold grows as a saprophyte on dead plant material in all forest nursery seedbeds in the Pacific Northwest. Under optimum conditions, it causes significant damage to densely grown bareroot seedlings. The disease can also be particularly damaging to container stock, both in the greenhouse and

**Gray mold may be confused with:**

- Frost damage**
- Rosellinia needle blight**
- Senescent needles**

outdoors when seedlings are set out to harden off. *Botrytis cinerea* may also ruin packed nursery stock under poor storage conditions where daily temperatures fluctuate from the ideal—near freezing—to 10 or 15 degrees higher. As damaging as gray mold can be, however, it responds quickly to changes in microclimate and management practices, and therefore is easily controlled in most cases.



**Figure 4-1.** Dead lower needles caused by *Botrytis* on densely sown Douglas-fir seedlings.

## Disease and hosts

Western hemlock, Douglas-fir, and Sitka and Engelmann spruce can all be severely damaged by gray mold. Though pines are somewhat resistant, probably because of their more open growth habit, they are susceptible under conducive conditions. True firs fit in the same susceptibility category as the pines. In California, gray mold is especially damaging to giant sequoia and coastal redwood. Hardwoods can also become infected. It is reasonably safe to assume that all forest nursery stock in the Pacific Northwest is susceptible.

Under the mild-climate conditions in nurseries west of the Cascades,

infection can occur at nearly any time. Gray mold often appears after an abiotic event that causes plants to die back, such as a winter freeze or spring frost, fertilizer burn, herbicide injury, or lower needle dieback in dense seedling stands. Such an event predisposes seedlings to infection by causing abnormal amounts of dead plant material upon which the disease builds.

A typical scenario in a coastal nursery would be a severe early-winter or spring freeze that kills many needles and portions of stems on seedlings. The warming, wet weather that usually follows a cold snap allows rapid and massive build-up of *Botrytis* inoculum on the dead plant material, creating a high

potential for infection of living plant tissue (See Figure 27-3). The sublethal plant injury causes leakage of cellular nutrients onto needle surfaces, stimulating spores that have landed there to germinate.

## Symptoms

Initial signs of gray mold on conifer nursery stock are usually found on the lower stems of seedlings in dense seedbeds or wherever

**Gray mold symptoms appear:**  
**All ages**  
**Year-round**

abnormal amounts of dead plant tissue are found (Figure 4-1). At first, degenerating needles appear to be water-soaked, and eventually develop tan, mushy spots. After the disease intensifies on masses of killed needles, it spreads upward into healthy shoots. When shoots are attacked first, the disease may move downward. Brown, sunken areas called cankers, caused by the fungus growing in the phloem, girdle healthy stem tissue (Figure 4-2), causing the entire shoot or leader to collapse and hang down in a withered mass of dead needles. Small, dark, irregular resting or survival bodies called sclerotia are found in the cankers on plant material that has been infected for some time. Under optimum temperature and humidity conditions, webs of reddish-brown mycelium produce masses of gray spores resembling grapes on a vine. These dry spores are released in gray clouds when disturbed. Both mycelium and spore clusters are easily visible to the naked eye (Figure 4-3).

## Fungus biology

Since spores, mycelium, and the resistant sclerotia of *B. cinerea* are

almost always present on dead plant debris, infection can occur nearly year-round in the moist maritime climate of coastal nurseries. Interior nurseries with definite winter seasons would have the greatest potential for infection during late summer and fall. The fungus probably overwinters as sclerotia and mycelium in old plant debris. It enters healthy tissue indirectly via wounds and attached dead vegetation, or directly by penetrating intact plant surfaces that have adequate free surface moisture (Figure 4-4).

Early in the infection process of healthy needles or leaves, germ tubes from germinating spores swell to form holdfast structures, anchoring the spore and initial hyphae to plant surfaces. Hyphal strands containing enzymes and toxins grow from the holdfasts and either penetrate and degrade the cuticle and epidermal cells, or enter through stomates (Figure 4-5).

*Botrytis cinerea* has an unusually wide temperature range, from 0 to more than 25 degrees C with optimal growth and conidia production occurring between 20 and 22 degrees C. Spore germination is known to occur at temperatures as



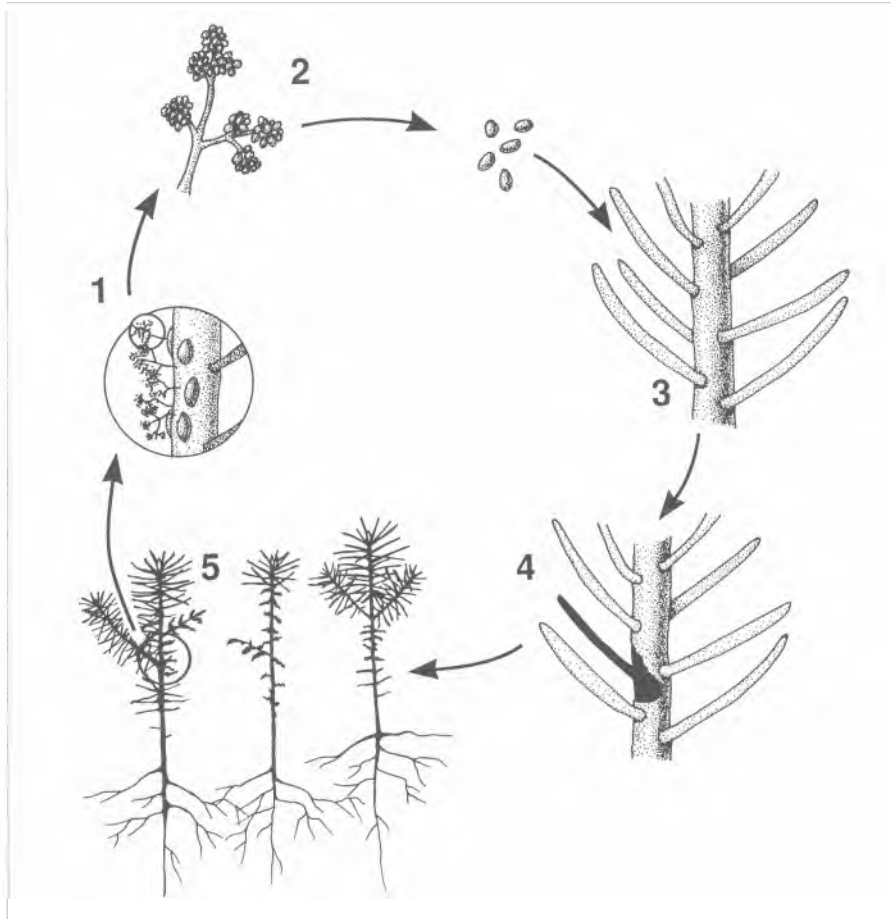
**Figure 4-2. Stem canker caused by *Botrytis*. Fungus first attacks needles and then grows from the dead needles into the stem. Photo courtesy of Tom Landis.**

low as 0 to 5 degrees C, which accounts for damage to trees in storage.

The conidial (asexual) state of another fungus, *Rosellinia needle*



**Figure 4-3. Close-up view of *Botrytis* sporulating on dead conifer needles.**



**Figure 4-4. Disease cycle of gray mold. The fungus overwinters on plant debris as mycelium or sclerotia (1), producing conidia which are transported by wind (2). Spores infect damaged, moribund, or dead needles (3), and the fungus may grow into the stem, sometimes causing a girdling canker (4). New conidiospores are formed on dead needles, spreading the disease to adjacent seedlings, particularly where planting density is high and moisture is retained on the lower, senescent foliage (5).**

blight, caused by *Rosellinia herpotrichiodes*, may be confused with *B. cinerea*. *Rosellinia* needle blight is often seen on the lower stems and branches of Douglas-fir seedlings under the same crowded conditions that bring on outbreaks of gray mold. The mycelium of the two fungus species are similar in appearance and color. Presence of the gourd-like *Rosellinia* perithecia (fruiting bodies) on the twigs of Douglas-fir seedlings distinguishes the two species. *Rosellinia herpotrichiodes*, however, is not common in coastal nurseries.

## Management

Gray mold can be largely eliminated with adjustments in cultural practices. It is essential to keep the microclimate within the canopy as dry and well aerated as possible. Also, better control of growth and hardening off allows seedlings and transplants to go into the dormant season in good health, reducing the amount of dead foliage in the event of an early freeze.

A significant change in general nursery management that resulted in reduced *B. cinerea* problems was

the lowering of seedling bed density from 50 to 25 trees per square foot. It was common for the more densely planted beds to have constant troubles with gray mold in the lower crowns. This management switch was initiated mainly to grow sturdier seedlings with better root systems, but there was an added benefit in the biological control of gray mold through lower humidity and vastly improved canopy aeration. Gray mold has never been a serious problem in transplant beds because they almost always have adequate air circulation.

Irrigation practices can be adjusted to help prevent infection. Normally, irrigation is either reduced or eliminated late in the season when the disease is at its most infectious state. Should an outbreak coincide with irrigation needs, it is important to make sure the foliage is allowed to dry quickly. Consider watering early in the morning when humidity is highest. Since free water is usually already present on foliage at this time, more water won't increase wetness, and midday winds can dry the foliage before nightfall.

Canadian researchers have found that 3 hours of temperatures around 15-20 degrees C and 98 percent relative humidity is sufficient for infection if there is free water on needle surfaces. The most recent research makes use of "needle wetness sensors" to obtain the most precise readings to date of optimum conditions for infection. Needle surface wetness offers a more direct measure of the conditions for successful infection than does relative air humidity. The sensor is connected to a data logger, which immediately informs the nursery manager when conditions are right for infection. This technology was developed primarily for container nurseries, where gray mold is an ongoing problem, but it could be used in bareroot nurseries as well. Removing diseased seedlings, particularly when diseased areas are small and localized, can reduce the spread of gray mold within a



**Figure 4-5.** Magnified needle showing hyphae of *Botrytis* originating from a spore, entering a stoma. Photo courtesy of Frank Dugan.

seedbed. This practice, however, may be economically unfeasible.

Fungicides are occasionally needed to control infections in dense foliage, in either nursery beds or containers. Fungicides may be used as protectants, to prevent disease fungi from taking hold after a severe freeze when large amounts of dead needles are produced. Once gray mold becomes entrenched in a crop, regular applications of approved fungicides may be necessary. Most fungicides do not cure the disease, but instead act as a protective spray by keeping it well below damaging levels. It is most important when applying fungicides to make sure the proper dose reaches the target. Spray nozzles must be aimed so that the spray penetrates the foliage, since gray mold grows mostly inside the canopy.

An effective standard fungicide mix for preventing crop-damaging outbreaks is captan and benomyl, 8 ounces of each in 50 to 100 gallons of water per acre. Chlorothalonil at 2 to 4 pints per acre in 100 gallons of water is equally effective. These fungicides can be applied at 10- to 14-day intervals as required. It is a

good idea to alternate them because of the tendency of gray mold fungus to build up resistance to a particular fungicide used exclusively. Other fungicides are also registered for control of gray mold.

Recommendations in the past called for dipping or spraying infected seedlings with a fungicide prior to packing. However, since human health risk has become a major factor in crop production and handling of pesticides, nursery managers should avoid using fungicides in close proximity to nursery workers.

Conifer seedlings in plastic-lined paper bags can be severely damaged by *B. cinerea* under poor storage conditions. However, handling practices in getting the trees from nursery bed to plantation have so improved over the last 2 decades that gray mold problems have been dramatically reduced.

Improved cultural practices, sanitary lifting, and cull procedures in the packing house keep dead needles and plant material out of the tree bags. Trees leaving nurseries today have such low moisture stress that bags would usually not need to be rewetted before planting. Such

trees will naturally be more resistant to gray mold infection during the trip from packing to planting. Gray mold problems can also be minimized by following the handling instructions that accompany nursery stock. See Chapter 8, Storage Molds, for a more complete discussion.

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## CHAPTER FIVE

# Lower Stem Canker

**Philip B. Hamm**

## Disease and hosts

Lower stem canker is caused by the fungi *Fusarium roseum* and *Phoma eupyrena*. It occurs primarily on Douglas-fir, although it is also found on noble fir. The disease has been reported in bareroot nurseries in western Oregon and Washington. Infection begins some time after October on 1+0 or fall-transplanted plug +1 seedlings. Symptoms usually do not develop until spring. Lower stem canker appears to be the same disease described by Morgan as *Fusarium* stem rot of Douglas-fir in Oregon. However, it is different from *Phoma* blight (see Chapter 15), which is caused by *P. eupyrena* on true fir. Both these fungi are widely distributed in agricultural soils.

**Lower stem canker may be confused with:**  
**Pesticide damage**  
**Phoma blight**  
**Phytophthora root rot**  
**Upper stem canker**  
**Winter desiccation**

## Symptoms

Symptoms first appear on seedlings in the beginning of their second year, in late winter or early spring as temperatures begin to rise. Needles encased in soil turn brown (Figure 5-1). Then tops of infected seedlings turn yellow, and later, reddish-brown. A canker—a sunken



**Figure 5-1.** Early symptom of lower stem canker. Lower needles encased in soil turn brown just before bud break.

region of dead tissue—appears on the stem, usually at the site of a needle scar or a bud. The canker enlarges until the stem is girdled. The seedling wilts rapidly if girdling occurs after new growth is present (Figure 5-2). The disease generally occurs in distinct areas within the nursery where drainage is poor. It spreads along the length of seedbeds more rapidly than across the beds (Figure 5-3).

Damage is often particularly heavy in areas where repeated tractor use during wet weather has formed large, deep puddles between beds. Muddy water splashed from these puddles, along with splashed rain, causes soil to build up on the stems of seedlings, creating what are termed soil collars. Soil collars are always present on infected seed-

lings. The girdling canker is found under the soil collar (Figure 5-4), often near the lower branches.

The foliar symptoms of lower stem canker are similar to those of *Phytophthora* root rot. However, lower stem canker can be recognized by the presence of the distinct girdling canker on the lower stem and by the absence of root decay.

## Fungus biology

The two organisms that cause lower stem canker may act singly or together. *Fusarium roseum* is most commonly associated with the disease, whereas *Phoma eupyrena* may or may not be present. Both organisms form resistant chlamydo-spores, thick-walled resting struc-

tures that survive in the soil. Inoculum comes from these chlamydospores or from conidia brought in on contaminated equipment, blown soil, or possibly seed.

Infection occurs beneath soil collars—apparently a favorable environment for survival and germination of fungi. Large numbers of *Fusarium* and *Phoma* propagules have been isolated from soil collars of symptomatic seedlings. Spores germinate and infect needles, buds, and stem tissue lying beneath the soil collar. Once they have infected the main stem, these fungi readily form a girdling canker.

**Lower stem canker  
symptoms appear:  
2+0, Plug + 1  
Early spring**

## Loss potential

Lower stem canker was responsible for killing millions of bareroot seedlings in nurseries throughout the Pacific Northwest during 1980 and 1981. Mortality in subsequent years has been reduced, but significant losses occur yearly in nurseries in the region. Mortality from lower stem canker in three nurseries caused average losses of 4 percent and 3 percent of 2+0 seedlings during 1983 and 1984, respectively. Transplanted plug seedlings are also particularly susceptible, perhaps because of the succulent nature of such seedlings, combined with transplanting stress, soil buildup on stems from transplanting and subsequent irrigation, and transplanting in nonfumigated ground containing large populations of *Phoma* and *Fusarium*.

## Management

### CULTURAL

Reducing the buildup of soil collars on 1+0 or plug +1 seedlings is



**Figure 5-2.** Wilting of new flush because of a girdling canker beneath the soil collar on the lower stem.



**Figure 5-3.** Area of heavy damage from lower stem canker. The disease spreads lengthwise along beds more rapidly than across them.



**Figure 5-4. Encased soil was removed from this symptomatic seedling, then the cortex was lightly scraped. Brown (necrotic) cambial tissue of the canker is clearly seen.**

essential. This can be done by mulching or by allowing moss to grow on seedbeds by avoiding some types of herbicides. Both straw dust and a mixture of redwood bark and sawdust have been shown to prevent soil collars when applied to nursery beds at a depth of 2-3 inches before heavy fall rains begin. Tractor use should be limited in the winter, particularly in areas where puddles form, to prevent splashing of soil and water onto seedlings. Maintaining well-sloped beds will also prevent wet areas.

#### **CHEMICAL**

Many chemicals have been tested for control of this disease but none have significantly lowered mortality. This is probably because the chemicals do not adequately penetrate soil collars and because there is no systemic fungicide that can move from foliage to tissues beneath soil collars.

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## CHAPTER SIX

# Phytophthora Root Rot

Everett M. Hansen

## Disease and hosts

Phytophthora root rot is caused by any of several *Phytophthora* species in Northwest nurseries. *Phytophthora megasperma* and *P. pseudotsugae* are the most common; *P. cactorum*, *P. cryptogea*, and *P. gonapodyides* are also regularly encountered. Two or more species may be present in a single nursery.

All these *Phytophthora* species have a wide range of hosts. Douglas-fir and the true firs are damaged in bareroot nurseries. Western and mountain hemlocks and sugar pine are also susceptible in inoculation tests and are likely to be damaged in the nursery if grown in high-hazard areas—places with poor drainage or a history of *Phytophthora* infestation. Western redcedar and incense cedar,

**Phytophthora root rot may be confused with:**  
**Mechanical damage**  
**Nematode damage**  
**Other root diseases**  
**Soil compaction**

lodgepole and ponderosa pine, western larch, and Sitka spruce sustain very little damage from *Phytophthora*. Port-Orford-cedar is resistant to most *Phytophthora* species, but is extremely susceptible to *P. lateralis*, which in fact attacks only Port-Orford-cedar.

*Phytophthora* species are water molds; they produce swimming zoospores in water-saturated soils.



**Figure 6-1. Root symptoms characteristic of *Phytophthora* root rot. Seedlings lack normal number and length of lateral roots, particularly in the lower areas, and the normally white cambial tissue becomes reddish-brown as the fungus advances upward.**

They infect trees at any time between germination and lifting as 2+0 or 2+1 stock. Seedlings are most likely to be damaged during the winter after seeds are sown or in the season following transplanting. *Phytophthora* species attack the fine roots of seedlings of all ages wherever soils are saturated. The frequency of infection increases with the population of the fungus. Above-ground symptoms are seldom visible before the end of the first growing season. *Phytophthora* species can cause damping-off of newly germinated seedlings, but at most nurseries

*Fusarium* and *Pythium* species are more damaging at this stage.

## Symptoms

### 1+0 AND 2+0 SEEDLINGS

In the first year root symptoms are normally confined to the lower one-third of the root system. Infected root tissue has a characteristic reddish-brown discoloration (Figure 6-1). There are usually no above-ground symptoms. Infected seedlings are most often confined to low-lying, poorly drained areas, but the infection may spread over large areas where heavy clay soils restrict drainage.

Above-ground symptoms generally appear in the spring of the second year. They consist of chlorosis, stunting, wilting, and browning of needles, followed by mortality. Branches show a stunted or "bottle-brush" appearance; this is due to delayed bud break and reduced elongation of branches and needles. Foliage symptoms follow a progression from chlorosis of the current year's needles and stunting of the shoots early in the growing season, to wilting and mortality by mid-summer (Figure 6-2).

Root symptoms include discoloration of the cambium, root decay, and finally the absence of roots. If cortical tissue (bark) is carefully scraped from infected roots with a knife, red to dark brown discoloration of the cambium will be evident. The border between healthy and infected root tissue is usually distinct but sometimes irregular, with fine streaks of discoloration penetrating





**Figure 6-2.** Progression of symptoms of *Phytophthora* root rot. Healthy seedling at left is followed by a chlorotic seedling with poor branch elongation and root development. The third seedling shows wilting and browning of current year's needle flush. The fourth seedling has been killed.

the healthy areas of the root. In severely infected seedlings, the entire root system may be dead. In addition, most secondary roots may be missing and the border between healthy and infected tissue will be located at or above the root collar.

Symptomatic seedlings most frequently are found where soil is poorly drained—in low spots, bed ends, or drainage contours; in heavy clay soils or those with high water tables; and in areas that have been flooded (Figures 6-3 and 6-4).

#### TRANSPLANTS

Above-ground symptoms generally are not seen in fall-transplanted seedlings until late winter or early spring. In seedlings transplanted in the spring, symptoms appear in late spring or early summer. Lateral roots and taproots of severely infected seedlings are stubby and have no fine roots. Occasionally, new shoot growth begins only to wilt quickly and die. Generally, transplants are extremely susceptible as a crop type, presumably because of root-pruning and transplanting stresses and because large amounts

of water are added after transplanting to prevent moisture stress.

The time symptoms appear and the distribution of affected seedlings in a field depend on whether seedlings were infected before or after

transplanting. Seedlings infected before transplanting will show symptoms earlier and symptomatic seedlings will be more evenly distributed throughout the field, although they will be more prevalent in wet areas. Trees infected after transplanting will show symptoms somewhat later and will usually be confined to wet or compacted areas.

#### Fungus biology

Seedling roots are infected by motile zoospores (asexual swimming spores), which are attracted to root tips and wounds (Figure 6-5). Zoospores germinate and their hyphae penetrate the root epidermis and grow within phloem tissue. Infected tissue is killed. Growth of the fungus is usually restricted to the phloem below the root collar; above-ground stem tissue quickly dries out after the roots die and thus cannot support the fungus. Zoospores can move short distances (a few millimeters) in saturated soil and much longer distances in water flowing over land.



**Figure 6-3.** *Phytophthora* root rot most often occurs in low areas where surface or underground water can accumulate, as shown by this area of low stand density in a 2+0 bed. Seedlings not killed have developed poorly.

Oospores are formed in killed seedling tissue by some *Phytophthora* species (Figure 6-6). They are the result of sexual recombination (meiosis). Their role in the infection process is not known, but probably is similar to that of chlamydospores, which are formed by some species. Chlamydospores (thick-walled, asexual resting spores) and oospores can withstand relatively dry conditions, enabling the fungus to survive throughout the summer when soil temperature is high and moisture is low. Presumably both oospores and chlamydospores can produce sporangia, which release zoospores under conditions of abundant moisture (Figure 6-7). Chlamydospores and oospores are moved in soil and on the roots of seedlings, as well as on vehicles and other equipment.

Roots generally become infected when the soil is moist enough for zoospore production and movement. In Pacific Northwest nurseries these conditions exist in fall, after frequent rains begin, and in spring through mid-summer, before nurseries reduce irrigation. In winter, when the ground is cold or frozen, activity of *Phytophthora* probably is quite low; little or no new infection occurs.

## Loss potential

*Phytophthora* root rot can ruin a nursery if it is ignored. Nursery blocks and even entire nurseries have been abandoned after infestation by *Phytophthora* species. Once established, the fungus is difficult, perhaps impossible, to eradicate. With careful disease management, however, and with special attention to maintaining good drainage, vigorous seedlings can be grown even in the presence of the fungus.

Where the disease is severe, many seedlings are killed outright, and more are infected to varying degrees even though they are still green at lifting. If lifting and storage conditions are not optimal, *Phytophthora* can spread throughout an entire bag of trees. Living but

infected trees show reduced survival rates after outplanting, although the fungus itself does not develop further once seedlings are planted in forest soils. Seedlings that have sufficient healthy roots to become established in the first growing season will not be further damaged by *Phytophthora*.

**Phytophthora root rot  
symptoms appear:  
1+0  
Summer  
2+0, transplants  
Late spring through late fall**

## Management

*Phytophthora* proliferates in saturated soils. Control strategies must be based on soil management that ensures good drainage. Fungicides are available to supplement cultural practices.

### CULTURAL

If possible, nurseries should be located on light rather than heavy

soils. Drainage can be improved by installing subsurface drainage systems and ditches to drain surface water, sloping fields towards drainage ways, subsoiling to break up hardpans, and building raised nursery beds.

If areas of a nursery or field cannot be adequately drained, then seedlings susceptible to *Phytophthora* should not be sown or transplanted in those areas. Growing such seedlings in chronic wet spots will result in large-scale losses or unsalable seedlings, or both, year after year.

Only resistant or tolerant tree species should be grown in high-risk areas—places where *Phytophthora* was present in the previous crop, or low, poorly drained, or flood-prone spots. Pathogenicity trials and nursery observations have led to this categorization of the resistance of species:

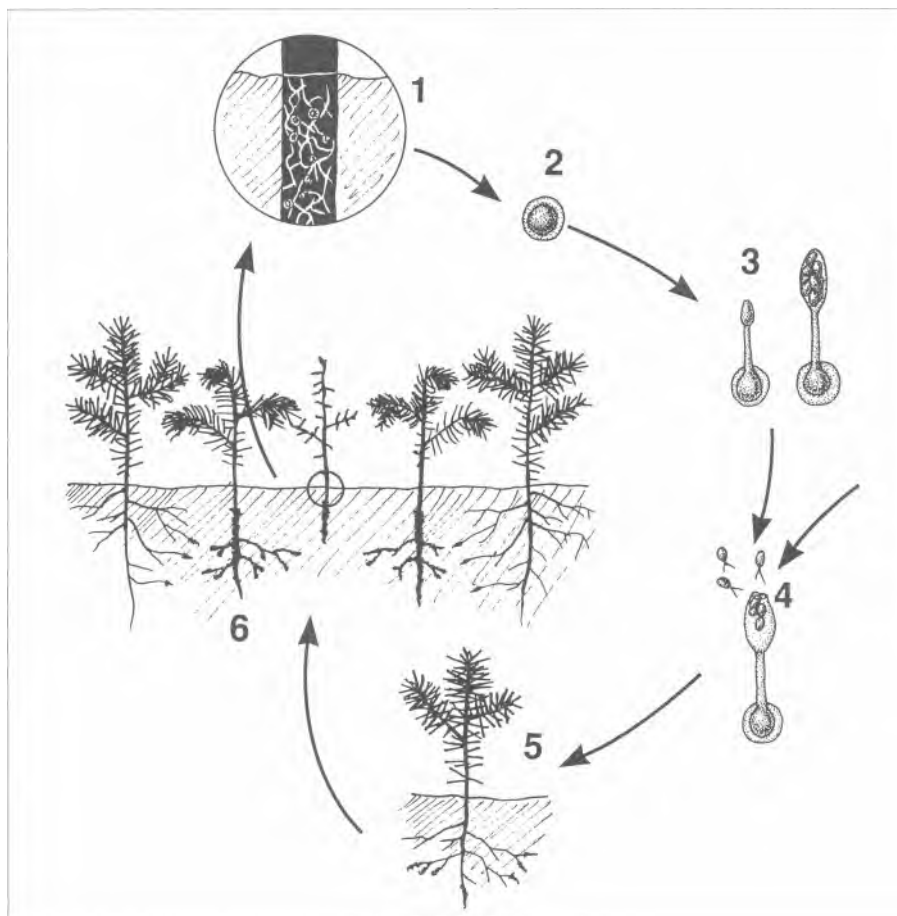
**Resistant** western redcedar

**Tolerant:** pine, spruce, larch, incense cedar

**Highly susceptible:** western and mountain hemlock, Douglas-fir, true firs



**Figure 6-4.** A distinct diseased area is defined by necrotic and chlorotic seedlings in rows where water has accumulated after transplanting.



**Figure 6-5.** Life cycle of *Phytophthora*. Decomposition of dead roots (1) leaves chlamydo-spores or oospores (2) in soil and host tissue. Spores germinate during favorable periods, producing sporangia (3); zoospores are released (4) and migrate short distances through saturated soils or long distances in flowing surface water to infect seedling roots (5). Infected tissue is killed. More zoospores are produced on freshly killed tissue; they infect nearby seedlings (6).

Irrigation practices can be tailored to keep soils well drained. Routine irrigation schedules should be modified as extra water is applied for cooling, frost protection, fertilization, and pesticide treatments. Over-irrigation and periodic flooding of soils should be avoided, particularly in areas known to be infested or into which infected seedlings are to be transplanted.

Patches of diseased seedlings should be lifted separately from the main crop and disposed of. Removing infected root and stem tissue removes inoculum from the soil and prevents the mixing of diseased and healthy seedlings. Rogued seedlings and packing-house culls should not

be returned to fields as organic matter or mulch because chlamydo-spores and oospores can survive in seedling tissues. Movement of stock between nurseries should be kept to a minimum to avoid initial or further *Phytophthora* contamination. Finally, careful observation of seedlings—particularly their root systems—will encourage early recognition and more-effective disease control.

#### CHEMICAL

Chemical soil and water treatments provide an important supplement to cultural control of *Phytophthora*. It is important to recognize, however, that they cannot replace

proper soil and water management.

**Fumigants**—Fumigants significantly reduce soil populations of *Phytophthora*, but do not always prevent diseases caused by these fungi. The fumigant may not penetrate heavy, poorly drained soils sufficiently to kill all the spores. In addition, *Phytophthora* may easily be reintroduced accidentally into fumigated beds by means of contaminated water, soil, or seedlings.

**Fungicides**—Several fungicides are registered for use against *Phytophthora* root rot on conifers and woody ornamentals, but only metalaxyl has been shown in extensive field tests to be effective on conifers. Applying metalaxyl to *Phytophthora*-infected seedlings will reduce their mortality and lessen the severity of the disease.

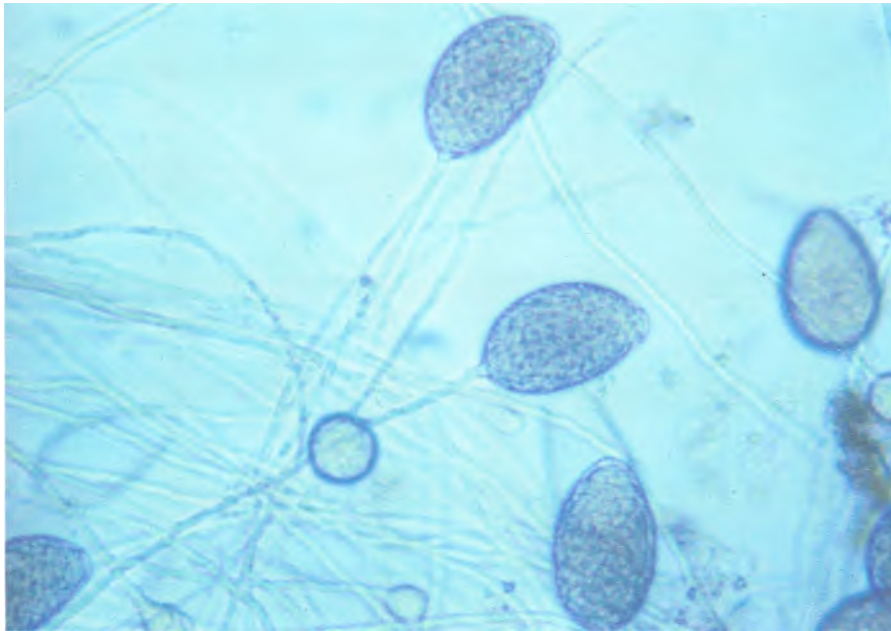
Metalaxyl is a systemic fungicide that can be applied to the soil in liquid or granular form. The fungicide is taken up from the soil by the roots. It halts the progression of the fungus through the root system; however, *Phytophthora* will remain viable in roots. Metalaxyl inhibits the formation of sporangia, oospores, and chlamydo-spores, and to a lesser extent mycelial growth and chlamydo-spore germination. It is also active against *Pythium*, but has little or no effect on other fungi. It does not harm mycorrhizae.

Metalaxyl must be used with discretion for two reasons: frequent use may cause naturally occurring tolerant strains of *Phytophthora* to become predominant, and repeated use in other crops has caused the buildup of organisms in the soil that quicken the breakdown of the chemical. The presence of *Phytophthora* in symptomatic seedlings should be confirmed by a pathologist before treatment.

One well-timed treatment of metalaxyl per year appears to adequately control the progression of *Phytophthora* root rot. The best times to treat seedlings are spring or fall when roots are active and can take up the fungicide. The fungicide is retained by the plant for a longer period during those seasons because



**Figure 6-6.** A *Phytophthora* oospore, microscopic in size, is thick-walled and therefore resistant to drying. With sufficient moisture and proper temperature, this structure will germinate to form a sporangium.



**Figure 6-7.** Sporangia produced by *Phytophthora*. In water, these structures each release many zoospores, which can cause infection.

Irrigation water can be tested for the presence of *Phytophthora* propagules. If it is contaminated, the nursery should switch to another source if possible. If no other sources exist, water can be chlorinated at the nursery. Various types of chlorination systems are available and can be adapted to each nursery's needs.

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soil temperatures are relatively cool. Metalaxyl can also be used as a preventive treatment, but this should be done only when seedlings or transplants must be sown in areas of known *Phytophthora* infestation.

*Water chlorination*—Chlorination of contaminated irrigation water can help check the spread of the disease. Water from agricultural districts and canal systems is more likely to be contaminated with *Phytophthora* than well water or municipal water.

## CHAPTER SEVEN

# Seed Fungi

**Willis R. Littke**

Several different species of fungi are associated with conifer seed. Early studies linked declines in germination in conifer seedlots to high levels of seedborne mold fungi. However, such occurrences are sporadic and hard to predict. Cone handling and storage practices after harvest affect levels of seedborne fungi significantly.

Seedborne fungi also contribute to outbreaks of Fusarium and Sirococcus diseases in container and bareroot conifer nurseries. These problems are covered elsewhere in this book.

## Fungi and hosts

Seedborne fungi (Figure 7-1) are very common in conifers and other plant species. The literature contains numerous references to fungi isolated from cone integuments (bracts and scales), seed coats, and embryos. The seedborne fungi of Douglas-fir and ponderosa pine have been studied more thoroughly than those of other Pacific Northwest timber species. Some of the more common fungi are listed in Table 7-1 on page 24.

## Fungus biology

The process through which fungi become established on Douglas-fir seed has been well documented. Less information is available for other tree species, but similar circumstances surrounding the buildup of inoculum on cones and its subsequent transfer to seed can be inferred. Fungal infection of seed

before the ripening of the cones appears to be minimal. Exterior portions of the cone may have a high degree of fungal association, even though interior infection levels may be low. The loose association of the inoculum with the cone surface strongly suggests that airborne spores are deposited there.

Once spores are deposited on seeds in open ripe cones or trans-

ferred from cone surfaces to the seed, they remain on the seed in a dormant state until environmental conditions are conducive to their germination. The opportunity for spores to germinate and infect seed can occur many times between cone harvest and sowing of seed, such as in cone bags when humidity and temperature build up or during stratification when seeds are kept moist. Spread of fungi on cones or fungi already on seed to uncontaminated seed can occur during seed extraction, stratification, and sowing, as well as at other times.

**Seed fungi symptoms appear:**

**1+0**

**Late spring  
through early summer**



**Figure 7-1. Fungi growing from true fir seed in a germinator. The amount of fungal contamination can vary by tree species and method of collection, and between seedlots.**

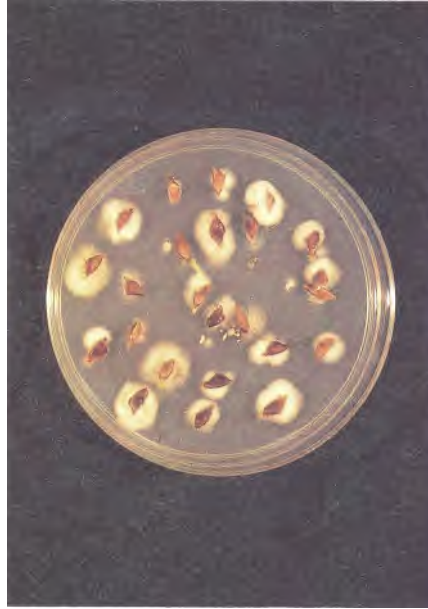
## Loss potential

It is difficult to predict damage from seedborne fungi. The most common fungi are saprophytic or even beneficial because they compete with other potentially pathogenic species. Some, however, are consistently associated with reduced germination rates and vigor. In general, fungi that are present within seed are more damaging than those that merely contaminate the outer seed coat. *Trichothecium*, for example, can reduce germination of Douglas-fir seedlots by 20 percent. *Caloscypha* is still more damaging; this fungus penetrates and kills seeds before germination. It can spread during cool, moist storage and even after sowing. Damage from *Caloscypha* has been most severe in British Columbia and in Europe.

## Management

Seed fungi are common in the environment and increase rapidly under warm, wet conditions. Preventing contamination is generally more successful than trying to cure diseased seedlots. Collecting clean cones, drying them, and extracting seed promptly are essential first steps. Storage of cones on the forest floor is important in the transmission of the seed fungus *Caloscypha*. Molds can develop in cone storage bags even with proper handling. Levels of seedborne fungi have been shown to be significantly higher following cone storage and extraction. This evidence might pinpoint the extraction phase as the primary stage when inoculum transfer occurs. Because cones appear to be a prime source of inoculum, the potential for cross-contamination between seedlots during cone processing is high.

Seed may be tested for the presence of fungal inoculum by plating seed on agar media. Testing should be selective enough to pinpoint the



**Figure 7-2. Douglas-fir seed in a petri plate. Notice that most seeds have white, cottony colonies of fungi growing from them.**

pathogen and to identify individual seedlots that need treatment. Fast-growing fungi and bacteria are readily isolated on non-selective media such as potato dextrose (PDA) or 2-percent malt extract (Figure 7-2). Selective or semi-selective media may be used to detect most seed fungi.

Detecting internal fungi requires sterilization of the seed surface with 1-percent bleach or 30-percent hydrogen peroxide, or a combination of the two. This will remove approximately 90 percent of the surface fungi. Better results can be achieved by increasing the soaking time, washing seed in a surfactant, and rinsing repeatedly in sterile distilled water.

Certain pathogens can be detected more quickly and accurately with monoclonal antibody tests in an enzyme-linked immunosorbent assay (ELISA). To date, ELISA tests are available for *Sirococcus* and *Caloscypha* on spruce seedlots.

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**Table 7-1. Common seedborne fungi in Pacific Northwest conifer species.**

| FUNGUS   | HOSTS                                    | APPEARANCE IN CULTURE   | DISEASE PROBLEMS                              |
|--|--|---|---|
| <i>Alternaria</i>                                | many conifers                            | gray-green to black; septate, club-shaped spores                                    | radicle and cotyledon disease of white spruce |
| <i>Caloscypha</i><br>(= <i>Geniculodendron</i> ) | pine, spruce<br>true fir,<br>Douglas-fir | beige, whitish gray, orange to pale yellowish brown; smooth, colorless round spores | seed rot, reduced germination                 |
| <i>Cladosporium</i>                              | many conifers                            | dark velvety green; pickle-shaped conidia   | reduced vigor of germination                  |
| <i>Fusarium</i>                                  | many conifers                            | white, salmon to pinkish-red; macroconidia sickle-shaped with 3-4 septations        | reduced germination disease in seedbeds       |
| <i>Penicillium</i>                               | many conifers                            | green to bluish-green; small, round conidia   | reduced vigor of germination                  |
| <i>Trichoderma</i>                               | many conifers                            | white turning to green, fast-growing; small, round conidia                          | none; this is usually a beneficial fungus     |
| <i>Trichothecium</i>                             | many conifers                            | pinkish, two-celled conidia   | reduced germination                           |

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## CHAPTER EIGHT

# Storage Molds

Everett M. Hansen

Various mold fungi commonly grow on seedlings that are stored between lifting and transplanting to the field or another nursery bed. All tree species are affected, and many different fungi have been implicated. Most fungi reported on moldy seedlings are saprophytes or weak parasites in nursery soils.

## Symptoms

Mold usually begins on older, senescent or dead needles in the lower crown of seedlings. The appearance of the mold mycelium depends on the species of fungus, but is often off-white to gray, forming thin webs between needles (Figure 8-1). The mold may spread throughout the entire foliage mass and is

occasionally seen on roots and stems as well. The discovery of moldy seedlings is alarming, but often the fungi cause less damage than appearances would seem to indicate. Mold development does point to poor storage conditions, however, and these may induce physiological changes that reduce the growth capacity of seedlings.

**Storage mold may be  
confused with:  
Mycorrhizae**

Occasionally mold fungi colonize and kill healthy needles and even

stems. A simple "fingernail test"—a scraping of the bark on the stem of a seedling—quickly reveals the extent of damage. Bark that is soft, watery, and discolored is probably dead. When needles are colonized but stems and buds remain healthy, seedlings will generally survive, although first-season growth may be affected. Mold observed on roots is most commonly from beneficial mycorrhizal fungi and is no cause for concern. However, root pathogens such as *Pythium* and *Phytophthora* can increase in storage and cause serious damage, especially if temperatures are above 3 degrees C (37 degrees F). Again, the fingernail test will quickly distinguish healthy from killed root tissue.



A



B



C

Figure 8-1. Dead foliage and stems caused by mold fungi that developed on Douglas-fir (A and B), and cedar (C) during storage.



## Management

Sanitation and storage environment are the two most important variables in controlling the development of mold. Seedlings already diseased in the nursery beds should be separated from healthy ones before packing. Trees infected with *Botrytis* or *Phytophthora* may contaminate whole bags of seedlings in storage. Soil is a primary source of mold fungi. Seedlings that are carelessly lifted and packed, with excess wet soil still clinging to roots and scattered through the foliage, are more likely to mold than clean seedlings.

Temperature and moisture are critical for the development of mold. Temperatures must be above freezing for fungi to grow, and growth increases dramatically as temperatures rise above 5 degrees C. Limited mold development may occur at lower temperatures, particularly after prolonged storage, but will seldom damage seedlings. Good temperature control, especially in densely packed lots of seedlings, is essential to maintaining the health of seedlings in storage. Mold fungi also need free water for spore germination and colonization of needles. Seedlings packed dry have less mold than wet seedlings, but it is difficult to dry seedlings and keep them dry without desiccating them during storage. It is best to pack seedlings moist to reduce water stress, and prevent mold development with strict control of temperature.

Freezer storage of seedlings eliminates the risk of mold development during the storage period, but seedlings are still at risk during the cooling-down and thawing processes. Continuous monitoring of seedling temperatures is important regardless of how seedlings are stored. Sensors should be placed in areas of least air circulation as well as in more-accessible locations to assure that conditions are uniform.

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## CHAPTER NINE

# Upper Stem Canker

**Philip B. Hamm**

## Disease and hosts

Upper stem canker is caused by the fungi *Phoma eupyrena* and *Fusarium roseum*. It has been reported only on 1+0 Douglas-fir seedlings. However, inoculation tests indicate that true firs, spruce, hemlock, and western larch are susceptible. This disease has been found in nearly every bareroot nursery in western Oregon and Washington. The fungi are present in nurseries outside this area, but the disease has been observed only in the Pacific Northwest.

**Upper stem canker may be confused with:**

- Frost damage**
- Gray mold**
- Mechanical damage**
- Pesticide damage**
- Phomopsis canker**

## Symptoms

Seedling tops turn chlorotic and then reddish-brown above girdling cankers on the middle to upper stem (Figure 9-1). Cankers initially appear as sunken areas centered most often on a bark fissure, a wound on the stem that occurs naturally when the bark is expanding during periods of rapid growth (Figure 9-2). Less often the canker will be associated with a needle (Figure 9-3).

The canker turns reddish-brown as it expands around the stem. Light



**Figure 9-1.** This seedling was top-killed by a stem-girdling canker midway up the shoot.

scraping of the cortex clearly reveals the infected area. More than one canker may occur on a seedling. Cankers may elongate up the stem, but there is relatively little downward movement. Pycnidia, small black structures, may be present on the face of the canker (Figure 9-2).

Damage is first evident in mid-August to September of the first growing season. Symptoms of top kill generally continue to appear through November. However, seedlings infected late in the fall may not show symptoms until the following spring. Damage is often concentrated in distinct areas of the nursery, with seedlings in other areas remaining unaffected by the disease (Figure 9-4).



**Figure 9-2.** An infected bark fissure shows brown discoloration of the normally green stem and small black pycnidia forming in the center of the fissure.

## Fungus biology

*Phoma eupyrena* and *Fusarium roseum* are common soil inhabitants and are associated with a number of other seedling diseases. They are commonly present on healthy seedlings. Primary inoculum probably comes from chlamydo-spores, thick-walled resting structures surviving in the residue of previous crops or weeds; or from conidia. *Phoma* conidia are small oval-shaped spores that form in small, black, globose pycnidia, whereas *Fusarium* conidia are sickle-shaped and are produced on the surface of the seedling. Conidia are produced in large quantities by both fungi and presumably act as secondary inoculum. Spores are brought into the nursery by contaminated equipment, seed, and soil,

including windblown soil. Infection of seedlings occurs when spores land in bark fissures. Needles are also occasionally infected. Infection occurs in late summer or fall, with damage first visible in mid-August.

**Upper stem canker symptoms appear:**  
**1+0**  
**Late summer through late fall**

## Loss potential

Significant losses occur sporadically in Pacific Northwest forest nurseries. Millions of seedlings have been damaged by upper stem canker, particularly during the years 1980 through 1985, when as much as 3 to 4 percent of the 1+0 crop in the Pacific Northwest was infected annually. Losses have been less severe in recent years because cultural and chemical control has become more effective.

Although seedlings are usually not killed, they develop multiple stems, which may make them unacceptable to buyers. Also, infected seedlings may not regain



**Figure 9-3.** The canker is associated with the dead needle attached to the stem and the bark fissure.



**Figure 9-4.** Typical field symptoms of upper stem canker. Seedlings tops are killed, generally in distinct areas of the beds.

adequate height to make packing standards.

The amount of infection depends on whether seedlings are actively growing or dormant at the time favorable for infection, as well as the amount of rainfall at that time. Damage can be particularly heavy among succulent seedlings when 1+0 seedling growth continues into the fall. Cool, rainy conditions favor the dissemination and germination of the fungus, while slowing the callusing of bark fissures and thereby leaving wounds open for prolonged periods.

### Management

#### CULTURAL

Cultural practices that force growth into the fall, such as fertilization and irrigation, should be avoided. Historically, high-density sowings have suffered greatest damage. Sowing seed less densely may effect some control.

#### CHEMICAL

The fungicides benomyl and chlorothalonil control upper stem canker when applied at 2-week intervals from mid-July to bud set in the fall. Applying them alternately in

rotation is recommended. More-frequent applications in the fall are necessary if heavy rains begin before buds set and bark fissures callus over.

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CHAPTER TEN

# Charcoal Root Disease

**John T. Kliejunas**

Charcoal root disease is caused by the fungus *Macrophomina phaseolina*. It occurs primarily in forest tree nurseries located in warm, lowland agricultural areas. The disease is absent from cool, high-elevation, forested areas and from the cool areas of the Pacific Coast. It has caused major losses

**Charcoal root disease may be confused with:**  
**Fusarium hypocotyl rot**  
**Fusarium root rot**  
**June beetle larvae damage**  
**Nematode damage**



**Figure 10-1.** *Macrophomina phaseolina* infection on a sugar pine seedling. Root infection is spreading to the hypocotyl.



**Figure 10-2.** Microsclerotia of *Macrophomina phaseolina* under bark on the stem of a dead slash pine seedling. Photo courtesy of USDA Forest Service, Forest Pest Management, Region 8.

up to 50 percent—in some California nurseries, but has been reported only once in a nursery in Oregon. In the West, sugar pine, Douglas-fir, giant sequoia, and true firs are highly susceptible. Ponderosa and Jeffrey pines are somewhat resistant. In California, the fungus is active when soil temperatures exceed 15 degrees C. The disease intensifies during hot summer weather because of the increased fungal activity and increased stress on seedlings.

The root's cortical tissues become infected (Figure 10-1), causing necrosis and blackening of root tips, lateral roots, and the root crown. The plant becomes stunted and yellowed, and dies. Masses of small, black, spherical resting structures (microsclerotia), visible with a hand lens, are formed in the dead inner

bark of the roots and lower stem (Figure 10-2). The microsclerotia are released into the soil after the roots decay. They remain dormant until they come into contact with new roots, and then germinate to initiate new infections.

Besides those seedlings that are killed in seedbeds, losses may occur when plants are culled or when

infected seedlings are transplanted, particularly if transplanted into warm soils.

No effective fungicides are available. Fumigation of the soil with a mixture of methyl bromide and chloropicrin before sowing greatly reduces levels of dormant microsclerotia and helps to control the disease. Eliminating weed hosts will reduce inoculum in the soil.

**Charcoal root disease symptoms appear:**  
**1+0, 2+0**  
**Spring through fall**

## Selected references

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## CHAPTER ELEVEN

# Fusarium Root Necrosis

**Alan Kanaskie**

*Fusarium* root necrosis is caused by the fungus *Fusarium oxysporum*. It affects 2+0 bareroot Douglas-fir seedlings. To date it has been observed in only one nursery in southwestern Oregon. Above-ground symptoms first appear in the spring of the second year and include stunting, needle yellowing, and mortality (Figure 11-1). Infected seedlings show dead lateral roots and elongated dark brown or red-

**Fusarium root necrosis symptoms appear:**  
2+0  
Spring

dish-brown lesions on taproots (Figure 11-2). Infected roots are not decayed and can be distinguished from roots affected by *Phytophthora* root rot by identifying the causal

**Fusarium root necrosis may be confused with:**  
**Phytophthora root rot**

fungus in the laboratory. Damage is most severe near ends of beds and in wet areas. Leaving these areas unsown should lower the frequency of occurrence of the disease. Fungicides would likely be ineffective because they do not penetrate the soil well enough to protect roots.



**Figure 11-1. Field symptoms of *Fusarium* root necrosis on 2+0 Douglas-fir include necrosis, chlorosis, and stunting.**

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**Figure 11-2. Dead lateral roots and dark brown or reddish-brown lesion on taproot.**

## CHAPTER TWELVE

# Larch Needle Cast

**Robert L. James**

This disease is caused by the fungus *Meria laricis*. The pathogen infects western larch seedlings in nurseries in northwestern North America. Outbreaks of the disease have been reported in bareroot nurseries in Washington and Idaho. Larch needle cast appears to be especially damaging to 2+0 bareroot stock at the beginning of the second growing season.

**Larch needle cast may be confused with:**  
**Frost damage**  
**Gray mold**  
**Pesticide damage**

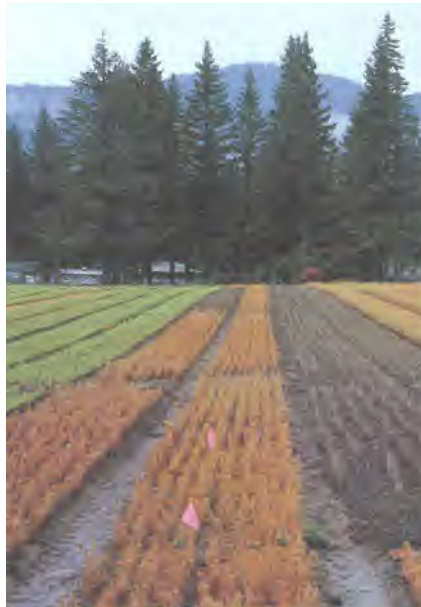
Symptoms appear as necrotic lesions on newly emerging needles formed shortly after bud break in the spring. The disease increases in severity as the season progresses, sometimes causing spectacular field symptoms (Figure 12-1). Severely infected needles are shed pre-

**Larch needle cast symptoms appear:**  
**2+0**  
**Late spring through early fall**

turally in the early summer (Figure 12-2). Although heavily infected seedlings may be killed, damage more commonly takes the form of

reduced height and stem caliper, which results in the culling of seedlings at lifting. Cool, wet weather in spring and early summer favors growth of the fungus.

The disease can be controlled in nurseries by application of chlorothalonil to the foliage of seedlings at 2-week intervals, beginning shortly after bud break and ending when warm, dry weather prevails. Damage may also be reduced by out-planting or transplanting seedlings after 1 year. This practice ensures that seedlings are not close to spore-producing needles that were shed during the first growing season.



**Figure 12-1. Damage to 2+0 western larch seedlings caused by larch needle cast. Green seedlings are non-host species.**

## Selected references

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**Figure 12-2. A close view of damage to needles of 2+0 western larch seedlings. Note cast needles on ground.**

## CHAPTER THIRTEEN

# Lophodermium Needle Cast of Scotch Pine

Alan Kanaskie

Lophodermium needle cast of Scotch pine (*Pinus sylvestris*) is caused by one or more fungi of the genus *Lophodermium*, including *L. seditiosum*, *L. staleyii*, and *L. pinastri*. Of these, *L. seditiosum* is the most damaging to seedlings. Infected needles show light green to

**Lophodermium needle cast may be confused with:**  
**Frost damage**  
**Nutrient problems**  
**Pesticide damage**

brown spots or bands in late spring or summer, then turn brown and drop from the seedling the following winter or spring (Figure 13-1). Shiny black fruiting bodies can be found on the brown needles (Figure 13-2). The disease can cause needle loss on large numbers of seedlings and may continue to affect trees after out-planting.

**Lophodermium needle cast symptoms appear:**  
**2+0**  
**Spring through fall**

The disease can be prevented by sowing seed from seedlots free of cone scales and debris and by taking care not to move infected stock between nurseries. Pine trees adjacent to the nursery can provide a



**Figure 13-1.** Beds of susceptible Scotch pine showing typical browning of needles. An unaffected bed of ponderosa pine is on the left.

source of *Lophodermium* spores. The fungicides maneb, chlorothalonil, and benomyl can effectively reduce damage from the disease, but because spore release periods differ among *Lophodermium* species, it may be necessary to apply fungicides monthly throughout the year.



**Figure 13-2.** Infected *Pinus contorta* needle showing dark fruiting bodies of *Lophodermium seditiosum*.

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## CHAPTER FOURTEEN

## Needle Rusts

Everett M. Hansen

Several needle rusts occur in conifer nurseries, but that caused by *Melampsora occidentalis* is the most common. Douglas-fir and western larch are the principal coniferous hosts, although pines may be infected as well. The fungus requires two hosts to complete its life cycle. Black cottonwood (*Populus trichocarpa*) and balsam poplar (*P. balsamifera*) are common alternate hosts in the West.

**Douglas-fir needle rust may be confused with:**  
**Frost damage**  
**Gray mold**  
**Nutrient problems**  
**Pesticide damage**

Symptoms on conifers are distinctive: yellow pustules filled with spores develop on the undersides of current-year needles in June or July (Figure 14-1A). These spores will

**Douglas-fir needle rust symptoms appear:**  
**All ages**  
**Summer**

infect nearby poplars but not other conifers. Conifers are infected in the spring shortly after bud break by spores formed on overwintered



A



B

**Figure 14-1. Melampsora needle rust: (A) aecia on the underside of Douglas-fir needles, and (B) uredia on the underside of a cottonwood leaf. Cottonwood is the alternate host.**

cottonwood leaves (Figure 14-1B). Heavily infected needles will drop off.

Melampsora needle rust is seldom damaging. However, infection can be reduced or eliminated by removing the alternate host from areas adjacent to nursery beds, by disposing of fallen leaves of the alternate host, and by applying fungicide to infected conifers. Chemicals (chlorothalonil, mancozeb, and maneb) should be applied during the 3-week period after bud break. Spray schedules depend on the compound selected.

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## CHAPTER FIFTEEN

# Phoma Blight

**John T. Kliejunas**

Phoma blight, caused by the soilborne fungus *Phoma eupyrena*, has caused severe needle loss of Douglas-fir and mortality of red and white fir in northern California. The

**Phoma blight may be confused with:**  
**Frost damage**  
**Lower stem canker**  
**Pesticide damage**  
**Winter desiccation**

fungus has also been associated with stem cankers of Douglas-fir in the Pacific Northwest and chlorosis of lodgepole pine seedlings, needle and twig blight of Engelmann spruce, and mortality of mugo pine. Tip death of ponderosa and lodgepole pine seedlings at nurseries in Montana, Idaho, and Oregon is associated with *Phoma* species and is covered in more detail in Chapter 18, Tip Blight of Pine.

**Phoma blight symptoms appear:**  
**2+0**  
**Winter through spring**

Phoma blight on Douglas-fir and true firs typically develops after heavy rains when seedlings are dormant between the first and second growing seasons. The splashing of muddy water against



**Figure 15-1.** *Phoma* infection of Douglas-fir needles ultimately causes a needle loss in the lower foliage.

the seedlings causes soil cones, or collars, to build up around stems and lower foliage. The fungus moves from the soil to invade needles and dormant buds. On Douglas-fir, symptoms first appear on the lower needles and spread upward. Needles turn chlorotic, then golden brown, and fall off (Figure 15-1). On true fir, dormant buds are infected, causing dieback or blight of terminal and lateral branches. The dieback starts at or near the buds and progresses down the stem. Seedlings die if all buds are killed (Figure 15-2). Tissues formed in the second growing season usually remain unaffected.

A preventive spray program in which chlorothalonil is applied at 2- to 4-week intervals during the dormant season (October to April) has re-



**Figure 15-2.** *Phoma* infection of red fir buds is followed by dieback of branches as the fungus moves down the stem.

duced the incidence of *Phoma* blight on Douglas-fir and true fir in northern California. Early sowing of seed increases the height of Douglas-fir seedlings during the first growing season. Foliage above the soil cones seldom becomes infected. Applying a mulch to nursery beds before the winter rainy season to reduce the splashing of soil has also reduced the incidence of the disease.

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CHAPTER SIXTEEN

# Phomopsis Canker

Everett M. Hansen

*Phomopsis lokoyae*, also known as *Diaporthe*, is one of several fungi that cause stem cankers on Douglas-fir seedlings. The diseases are most readily distinguished from one another by the ages of the seedlings they infect. *Phomopsis lokoyae* attacks seedlings in the 2+0 year, causing cankers at the base of new growth (Figure 16-1). This fungus is also occasionally associated with cankers on 1+0 seedlings.

Phomopsis canker is sporadic in most years in most nurseries, and

**Phomopsis canker may be confused with:**  
Frost damage  
Pesticide damage  
Upper stem canker

damage is usually negligible. Occasionally—most often following a long rainy spell at bud burst—the disease causes significant damage, with many seedlings in a nursery affected. The new shoot on an infected seedling is girdled and killed, but

**Phomopsis canker symptoms appear:**  
2+0  
Late spring through summer

since the fungus does not grow downward, lateral buds quickly replace the lost leader. Recent



**Figure 16-1. Diseased Douglas-fir seedling. Canker begins at or near bud scars, eventually girdling the stem and killing the top.**

research suggests that multiple-topped seedlings, whether caused by top pruning or Phomopsis canker, grow into trees of normal form in the field. Phomopsis canker is usually so sporadic that control is not warranted. When necessary, regular applications of fungicides (benomyl and chlorothalonil) reduce the incidence of Phomopsis canker.

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## CHAPTER SEVENTEEN

# Sirococcus Tip Blight

Jack R. Sutherland

*Sirococcus* tip blight is caused by the fungus *Sirococcus strobilinus*. In bareroot nurseries the disease has been recorded on white and Sitka spruce; lodgepole, Jeffrey, sugar, and ponderosa pine; and Douglas-fir. It occurs primarily in British Columbia, California, Idaho, and Montana nurseries, with infrequent occurrences reported in Oregon and Washington.

*Sirococcus* tip blight can affect up to 15 percent of any host species. Occurrence and losses are highest in nurseries with cool, moist, overcast conditions in mid- to late summer.

*Sirococcus* tip blight affects western hemlock in the forest but has not yet caused damage to that host in nurseries. The disease appears on bareroot seedlings from the last half of the first growing season to early in the second growing season.

*Sirococcus* tip blight affects random individual seedlings or small patches of seedlings. On all hosts the disease kills the current year's leader, usually from the tip downward (Figure 17-1). The desiccated tip of the terminal may assume a crozier shape. Foliage on the killed terminal dies from the base outward and turns straw-brown in color. Progression of the disease normally stops at the first one or two uppermost whorls of branches. The following spring one of the lateral branches turns upward as the leader. This may result in seedlings with multiple leaders.

Pycnidia form on the killed tissues, most abundantly at the bases of needles (Figure 17-2). They are initially butterscotch-brown, then dark brown at maturity. Spindle-shaped, multiseptate conidiospores ooze from pycnidia. They are the only known spores of *S. strobilinus*. Pycnidia also frequently form on old cones of various spruces and possibly pines. Conidiospores are disseminated to young, susceptible shoots of host seedlings via wind-borne water droplets or fog.

**Sirococcus tip blight may be confused with:**  
**Frost damage**  
**Pesticide damage**  
**Tip blight of pine**

Cool, moist, overcast conditions favor both infection by conidia and predisposition of the host to infection. Seedlings growing under low light intensity are more prone to infection. Once established in seedbeds, the disease can intensify by spreading to other seedlings. Irrigation further enhances the spread of *Sirococcus* and aggravates the conditions for infection.



A



B

**Figure 17-1.** *Sirococcus* tip blight on a 2+0 Douglas-fir (A) and a 2+0 lodgepole pine (B).



Figure 17-2. Pycnidia of *Sirococcus strobilinus* on killed Douglas-fir tissue.

The disease is rare in nurseries with dry, bright summer and fall days. Growing susceptible species in nurseries in drier areas might prevent losses from *Sirococcus* tip blight.

tered on windbreak trees or forest trees adjacent to the nursery. One outbreak of the disease in California was traced to inoculum from diseased cones that fell into nursery beds from nearby forest trees. It may be worthwhile to remove such trees if they are not too abundant or valuable.

**Sirococcus tip blight  
symptoms appear:**

**1+0**

**Late summer and fall**

**2+0**

**Late spring**

Fungicides such as chlorothalonil may be applied during the infection period. Applications should be frequent if rainfall is heavy, especially when seedlings are growing rapidly.

If the disease is controlled by fungicides, or if the diseased seedlings are removed from the nursery, the disease is not carried over from one seedling crop to the next. Thus each new disease outbreak must originate from inoculum overwin-

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## CHAPTER EIGHTEEN

# Tip Blight of Pine

Sally J. Campbell

Tip blight of lodgepole and ponderosa pine seedlings is a disease of unknown cause. *Phoma* species have been isolated, but their actual ability to cause tip dieback has not yet been demonstrated in pathogenicity tests. Tip blight has occurred at nurseries in Oregon, Idaho, and Montana. Death of the upper portion of the shoot is usually seen

Tip blight may be confused with:  
Frost damage  
Pesticide damage  
Sirococcus tip blight

in the spring and summer of the 1+0 or 2+0 year (Figure 18-1). Terminals and laterals may be affected. Killed shoots may form a crook, and needles on the killed stem die and turn brown. Multiple infections on the main stem and laterals can occur. These symptoms are similar to those caused by *Sirococcus strobilinus*, the cause of Sirococcus tip blight.

Tip blight symptoms appear:  
2+0  
Spring through summer

The incidence of disease increases after periods of high humidity. The



**Figure 18-1.** Seedling with upper portion of shoot killed. Infection of stem tissue results in a canker which eventually girdles the stem. Isolations from these stem cankers typically yield *Phoma* species.



**Figure 18-2.** Field losses due to tip blight of pine.

disease appears randomly throughout the seedbeds, although some seedlots have a much higher incidence of the disease than others (Figure 18-2). Losses are usually minor, with healthy laterals on many of the affected seedlings assuming dominance and growing sufficiently so that the seedlings are shippable at the end of the rotation. Mortality is rare.

Fungicides such as chlorothalonil may reduce the occurrence of tip blight if applied periodically in the spring and early summer of the growing season, beginning before the disease is seen. New infections decrease significantly with the onset of warm, dry weather; treatment is rarely necessary after mid-summer.

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## CHAPTER NINETEEN

# Western Gall Rust

Sally J. Campbell

## Disease and hosts

Western gall rust is caused by the fungus *Endocronartium* (*Peridermium*) *harknessii*. This fungus infects lodgepole and ponderosa pines as well as other two- and three-needle pines. It can be found in most North American bareroot nurseries that grow these hosts. The disease is most prevalent when susceptible pines adjacent to the nursery, such as those in windbreaks, are infected with the fungus.

Western gall rust may be confused with:  
**Mechanical damage**  
**Pesticide damage**



Figure 19-1. A 2+0 ponderosa pine showing swelling on the stem and branch proliferation from *Endocronartium harknessii* infection.

## Symptoms

Globose to pear-shaped swellings on the stem or branch of the seedling usually appear late in the second growing season (Figure 19-1). Galls continue to develop after outplanting and eventually girdle and kill the seedling stem or branch. Girdling is usually not seen in the nursery unless seedlings are grown for longer than 2 years. Similarly, spores are not produced on galls that develop in the nursery because the interval between infection and sporulation is greater than 2 years.

## Loss potential

The percentage of seedlings culled because of western gall rust infection is seldom greater than 1 percent of a

particular seedlot. However, a much higher percentage of infected but asymptomatic seedlings may actually be planted.

Western gall rust symptoms appear:  
**2+0**  
**Summer**

## Management

Because this rust does not cycle between pine and another plant species, removal of alternate hosts in the vicinity of the nursery is not a

control option. Risk of seedling infection can be reduced by removing infected branches or entire trees that are within 275 m (300 yards) of the perimeter of the nursery so that spores from mature galls do not blow into the nursery and infect seedlings. All seedlings with galls should be culled before packing to reduce the spread of the disease to outplanted areas.

No fungicides are registered in the Pacific Northwest specifically for western gall rust control. Other methods of control should be considered before fungicides are used.

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## CHAPTER TWENTY

# Cranberry Girdler; Sod Webworm

## *Chrysoteuchia topiaria*

David L. Overhulser

### Insect and hosts

The larval stages of the cranberry girdler commonly feed on the crowns and roots of grasses. Cranberry girdler feeding damage to the root-collar area of conifer seedlings has been reported in Oregon, Washington, and Idaho. Two-year-old Douglas-fir, noble fir, larch, and spruce stock are frequently damaged. Other stock types occasionally damaged are 1+0 and 3+0 Douglas-fir and 1+0 larch.

Cranberry girdler damage  
may be  
confused with:  
Lower stem canker  
Mechanical damage  
Rodent feeding  
Root weevil damage



Figure 20-1. Cranberry girdler damage to 2+0 noble fir seedlings. Oregon Department of Forestry photo.

### Symptoms

In late summer and early fall, cranberry girdler larvae eat patches of bark and cortex from the root collars and roots of seedlings (Figures 20-1 and 20-2). Damage is detected when stock is lifted and graded or when severely damaged seedlings still in the beds turn yellow in the fall.

### Insect biology

Adult moths (Figure 20-3) emerge from May to July in grass fields in or next to nurseries. Moths are visible during daylight; they fly in quick,

jerky movements for short distances. Female moths deposit eggs on and around nursery stock. Eggs hatch in 3 to 5 days. Larvae feed in nursery beds from June to October. The feeding of late-instar larvae from August to October causes the most damage to seedlings. In late fall, larvae spin a cocoon in the soil where they overwinter (Figure 20-4).

Cranberry girdler larvae and cocoons usually cannot be found by searching nursery soil. Moths are easily detected in the spring with traps baited with a commercially available attractant. Moth populations vary from year to year because

of the effects of predation and disease on overwintering larvae.

### Loss potential

Nurseries next to grass fields, which are prime cranberry girdler habitat, are most vulnerable. When the moth population is high, 15 to 20 percent of the seedlings in a single lot can suffer below-ground girdling. Since many foresters will not accept seedlings with cranberry girdler damage, the cull rate for infested seedlings can be high.



Figure 20-2. Cranberry girdler damage to 2+0 Douglas-fir. Note that callus has not formed around wounds, indicating that damage took place in late summer and fall. Oregon Department of Forestry photo.



Figure 20-3. Moth stage of cranberry girdler is present from May to July. Moths are generally 25 mm (1 inch) in length. U.S. Department of Agriculture photo.

## Management

### CULTURAL

Removing grasses in noncrop areas by cultivating or applying herbicides will eliminate prime habitat and therefore reduce infestation of nursery beds.

### BIOLOGICAL

Birds such as starlings, killdeer, sandpipers, and blackbirds feed on overwintering larvae. These birds are common in bareroot nurseries and grass fields. A naturally occurring soil fungus, *Beauveria bassiana*, also kills overwintering larvae.

### CHEMICAL

Traps placed in grassy areas next to the nursery will help in timing the applications of insecticides.

Diaz-inon, applied to nursery beds 3 and 5

**Cranberry girdler symptoms  
appear:  
2+0  
Late summer to lifting**

weeks after the male moths are detected in traps, is effective against ovipositing female moths. Applying chlorpyrifos to the soil in August and



Figure 20-4. Late-instar cranberry girdler larva in hibernaculum. Larvae are generally 25 mm (1 inch) in length. J.S. Department of Agriculture photo.

September kills small larvae feeding near the soil surface.

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## CHAPTER TWENTY-ONE

# Cutworms

## Noctuidae

David L. Overhulser

### Insect and hosts

Cutworm larvae feed on the foliage of a large variety of weeds and agricultural crops. Cutworm damage is usually a minor but chronic problem in bareroot nurseries throughout the Pacific Northwest. The cutworm species that damage 1+0 seedlings have not been precisely identified. Susceptible seedlings include Douglas-fir, true fir, pine, hemlock, spruce, and cedar.

**Cutworm damage may be confused with:**  
**Damping-off**  
**Fusarium hypocotyl rot**

### Symptoms

Damage is confined to young, succulent seedlings in the spring. The first sign of damage is cut or chewed needles (Figure 21-1). Then seedlings are clipped off at or near the ground line (Figure 21-2). Sometimes damaged seedlings develop an area of sunken stem tissue around a wound which resembles symptoms of damping-off (Figure 21-3). Other Lepidopteran larvae occasionally cause damage that resembles the feeding of cutworms.

Cutworm damage is frequently diagnosed without finding the insect. Cutworms normally feed at night and hide underground during the day, making them difficult to locate even in seedling beds with obvious damage. A typical cutworm is a dull-

colored, hairless larva, 2 to 50 mm in length (1/16 to 2 inches). Cutworms are generally sluggish if disturbed and often assume a characteristic curled position (Figure 21-4).

### Insect biology

Most cutworms that damage young seedlings have similar life cycles. Moths appear in late summer and early fall. Female moths deposit eggs on leafy plants or in the soil, where both eggs and larvae overwinter. Weeds in and around nursery beds provide prime cutworm habitat. In the spring,



**Figure 21-1.** Chewing on primary needles can be the first sign that cutworms are present in seedbeds.



**Figure 21-2.** As cutworm damage progresses, clumps of needleless stems appear in seedbeds.



**Figure 21-3.** Cutworm damage can cause sunken or depressed areas that resemble the symptoms of damping-off.

young larvae move into nursery seedbeds to feed. Cutworm larvae readily move from one site to another; this may explain their appearance even in seedbeds that were fumigated in the fall. Cutworm populations fluctuate greatly. Damage to seedlings can be high one year and nonexistent the next.

### Cutworm damage appears:

1+0

Late spring through fall

### Loss potential

Cutworm numbers are normally low in bareroot conifer nurseries, and their damage is usually not extensive. Cutworm feeding typically affects only a small portion of 1+0 beds at several locations within a nursery. In some years, however, cutworms are very abundant and widely distributed in seedbeds. Cutworms feed voraciously; a single larva can destroy many seedlings. Cutworm larvae also can be found

on the foliage of older seedlings, but their feeding does not cause significant damage.

### Management

#### CULTURAL

Aggressive weed control eliminates cutworm breeding sites within the nursery.

#### CHEMICAL

Insecticides such as diazinon and esfenvalerate are effective against cutworms. Beds of 1+0 seedlings should be examined weekly for damage symptoms for 6 to 8 weeks after germination. Areas with cutworm feeding should be marked for treatment. Since these areas are typically small, insecticides can often be applied with a small portable sprayer. Treatments applied in the late afternoon may be most effective because larvae are active only at night. Young, succulent seedlings are sensitive to chemical damage, so it is important to select insecticide formulations with a low risk of phytotoxicity.

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**Figure 21-4.** Typical cutworm larva in a curled position, with pupa. Mature larvae typically are 35 to 45 mm (1-3/8 to 1-7/8 inches) long.

## CHAPTER TWENTY-TWO

# June Beetle; White Grubs

## *Polyphylla* spp.

David L. Overhulser

### Insect and hosts

June beetles are common throughout the Pacific Coast states. The larvae, called white grubs, feed voraciously on the roots of a wide variety of ornamental and agricultural crops. Douglas-fir and true fir bareroot seedlings can be heavily damaged by white grubs, and most other conifer seedlings are probably also susceptible. Damage is often reported when conifer seedlings are grown in or transplanted into fields with sandy soil in which grass and weeds were previously growing.

**June beetle larvae damage**

**may be confused with:**

**Fusarium root rot**

**Mechanical damage**

**Root-lesion nematode damage**

**Root weevil damage**



**Figure 22-1. Noble fir seedlings in November with yellowing foliage from root damage caused by white grubs. USDA Forest Service photo.**

### Symptoms

Damage to seedlings normally occurs in late spring and summer. If it occurs late in the growing season, the seedlings may not turn yellow until fall or winter (Figure 22-1). Root damage is often so extensive that seedlings are easily pulled from the ground (Figure 22-2). White grubs are large enough to be visible to the unaided eye; they can often be located in the soil around damaged seedlings in the fall (Figure 22-3). Holes approximately 1 cm in diameter are sometimes visible on the soil surface. These are made by the

adult beetles when they emerge from the ground.

### Insect biology

The typical life cycle for June beetles in the Pacific Northwest takes 3 years to complete. Female beetles lay their eggs in June or July in the soil of grass fields or other areas with heavy vegetation. Eggs hatch in 3 to 4 weeks. The young larvae feed on decaying organic matter and fine roots. As winter approaches, larvae burrow 20 to 55 cm into the soil and remain inactive. During the spring months, larvae

move upward to feed on roots.

This pattern of seasonal migration in the soil is repeated the following year. Older larvae cause most of the damage to conifer seedlings. Pupation occurs in late May and June of the second year. Adult June beetles develop 6 to 8 weeks later but remain in the pupal cell beneath the soil until the following spring, when they emerge to mate and lay eggs. Adult beetles occasionally feed on conifer foliage, but cause negligible damage.

The immature and mature forms of June beetle are all quite distinctive



**Figure 22-2.** Nursery stock with extensive root damage from the feeding of white grubs. USDA Forest Service photo.

in appearance. Older white grubs, the type associated with the most extensive damage to seedlings, are approximately 2 to 5 cm long. These larvae are C-shaped, with a brown head and three prominent pairs of legs on the thorax. The adult beetle is 2 to 3.5 cm long and dull brown or metallic brown in color, with distinctive club-shaped antennae. Two common *Polyphylla* species have yellowish-white scales arranged in longitudinal stripes on their wing covers (Figure 22-4).

**June beetle larvae damage appears:**  
All ages  
Late spring through summer

### Loss potential

Seedling losses are greatest when stock is introduced into an area of light soils in which grass or weeds have been growing for 2 or more years. Under these conditions, even large seedlings can be severely damaged. Seedling losses of up to



**Figure 22-3.** White grub larvae have a characteristic C shape and three prominent pairs of legs. Larvae are typically 2 to 5 cm (7/8 to 2 inches) long. USDA Forest Service photo.

30 percent have been reported in British Columbia nurseries.

### Management

#### CULTURAL

Seedling beds with a history of continuous cropping rarely suffer damage from white grubs. Areas converted to nursery beds after being in sod for 2 or more years should be tilled several times in April to May or in September. Tilling or disking soils macerates grubs and exposes them to predators such as birds.



**Figure 22-4.** Adult female June beetles have large, club-like antennae. Beetles are 2 to 3.5 cm (7/8 to 1-1/2 inches) long. Oregon State University photo.

Cover crops can either encourage or discourage white grub infestations—grass supports white grub larvae, while legume crops do not.

#### CHEMICAL

Soil fumigation will eliminate white grub populations. Fumigation is recommended if seedlings are transplanted into formerly grassy or weedy areas. Drenching infested seedling beds with insecticides is not recommended because insecticides usually do not penetrate the soil well enough to kill the grubs.

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## CHAPTER TWENTY-THREE

# Lygus Bugs

## *Lygus hesperus*; *L. lineolaris*

Timothy D. Schowalter

### Insect pest and hosts

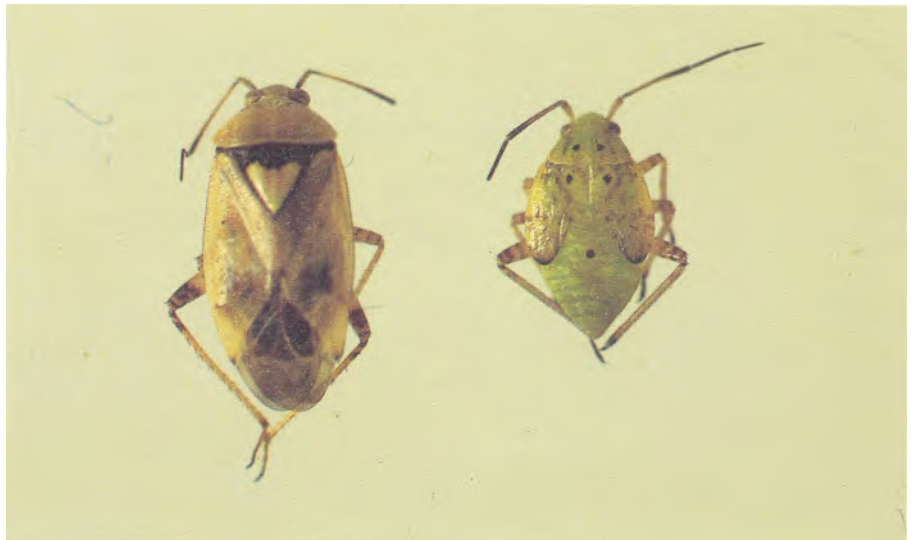
Lygus bugs (*Lygus hesperus* in Oregon, *L. lineolaris* in British Columbia) have become costly pests in conifer nurseries in the Pacific Northwest. Adults are 6 to 7 mm (1/4 inch) long; immatures (nymphs) are 1 to 6 mm long (Figure 23-1). Both can cause injury to seedlings.

Lygus bugs feed primarily on agricultural crops such as alfalfa, cotton, and fruit and vegetable crops, as well as a great variety of weeds, but they also feed on conifer seedlings. They appear to prefer pines over Douglas-fir and true firs, but they cause less damage to pines than to other conifer species.

**Lygus bug damage may be confused with:  
Pesticide damage**

### Symptoms

Seedlings are injured when the insect inserts its sucking stylet into growing shoots, typically the terminal shoot, and injects digestive enzymes to predigest the plant material. These enzymes and the host's physiological response to wounding cause a lesion to form around the wound (Figure 23-2). These lesions disrupt the growth of terminal shoots, causing them to be deformed (Figure 23-3). Loss of terminal dominance results in lateral shoot growth and, eventually, a bushy appearance.



**Figure 23-1.** *Lygus hesperus* adult (left) and nymph (right). Adults are 6 to 7 mm (1/4 inch) long; nymphs are 1 to 6 mm (up to 1/4 inch) long.

The bugs feed on 1+0, 1+1, and 2+0 seedlings. Injury occurs throughout the growing season, but most damage occurs when the bugs migrate after harvest of nearby alfalfa or other crops. In western Oregon, lygus bugs feed on 2+0 seedlings in the spring during shoot elongation, and then move onto 1+0 seedlings during summer and fall. Their move coincides with the germination and growth of these seedlings, the top pruning of the 2+0 seedlings, and the dispersal of the first generation of bugs (Figure 23-4). Bug damage resulting from late-season feeding is revealed the following spring as excessive lateral bud growth during shoot elongation.

### Insect biology

At least two generations of lygus bugs are produced annually in

western Oregon. Overwintering adults appear on 2+0 seedlings in May at bud break, and first-generation immatures appear soon after. Mature adults and second-generation immatures appear in July. Second-generation adults mature in August and overwinter.

Flightless nymphs are abundant throughout large blocks of seedlings, indicating that lygus bugs reproduce within nurseries. It is not clear whether the insects reproduce naturally on conifer seedlings or only on associated weed hosts such as groundsels and clovers. Spindle-shaped lygus eggs have been found on seedlings caged with adult lygus bugs, but nymphs hatched from these eggs have not developed successfully on seedlings.

## Loss potential

Susceptibility of seedlings depends on genotype, proximity of the nursery to other crops preferred by lygus, and size of the lygus population among those crops. Irrigation may make seedlings more susceptible to feeding injury because it stimulates new growth, which is preferred by the bugs.

**Lygus bug damage appears:**  
**1+0, 1+1, 2+0**  
**Spring through summer**

Cumulative damage in western Oregon has ranged from 10 to 50 percent. Early-season damage may be largely cosmetic, with seedlings reestablishing terminal dominance. Late-season damage is more severe, frequently resulting in death of



**Figure 23-2.** Lygus bug feeding injury to 1+0 Douglas-fir seedling. Note the lesion, up to 1 cm (3/8 inch) long, and deformed needles below the top of the seedling. Oregon Department of Forestry photo.



**Figure 23-3.** Seedling deformity resulting from lygus bug feeding (left), compared to an undamaged seedling (right). Note the lesion and needle deformity below the damaged top and the development of lateral dominance. Oregon Department of Forestry photo.

terminal buds and deformed growth in the following year. The long-term ability of damaged seedlings to recover height growth and resist future attack is unknown.

## Management

Lygus bugs can be detected by sweeping a fine-mesh net through seedling beds, by examining weeds in or near seedling beds for adults and nymphs, and by carefully checking seedlings for the first appearance of lesions and deformed tops.

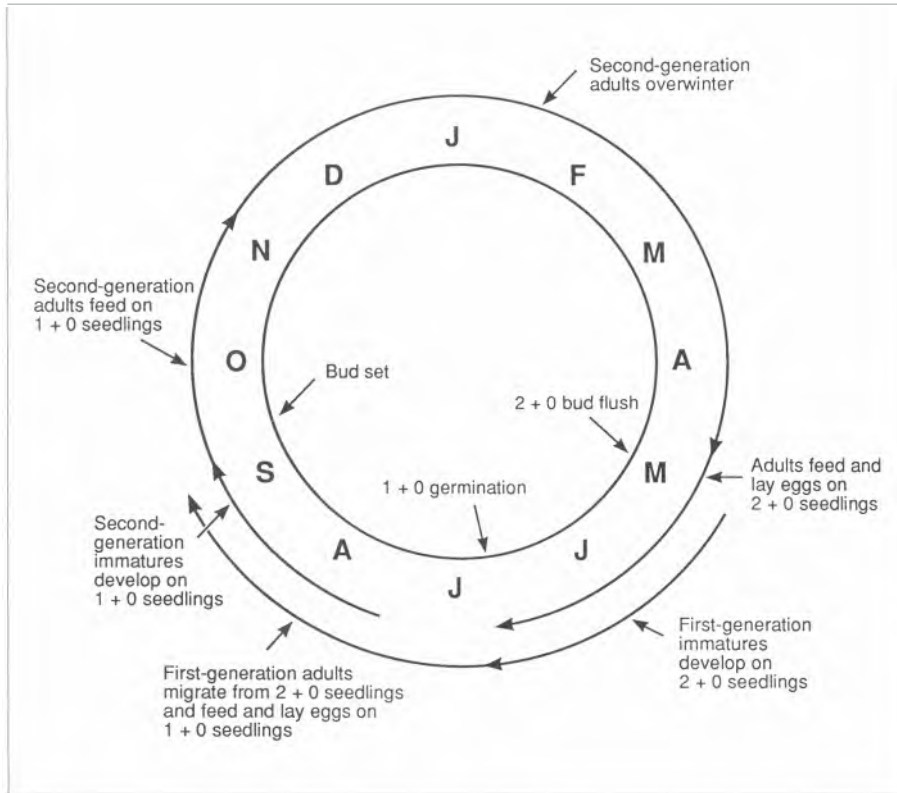
Pesticides may be applied at the first appearance of lygus bugs and periodically thereafter. Two to four applications of fenvalerate or acephate between mid-July (within 2 weeks of initial damage) and early September reduced damage by 80 to 90 percent. Later applications did not reduce the frequency of multiple tops in the following year. Pesticides should eliminate flightless nymphs. Control of the highly mobile adults requires spraying in the early morning when the insects are sluggish. Heavy irrigation dilutes

pesticides. Rapid seedling growth also reduces their efficiency. Alternatives to chemical control include isolating the nursery from alfalfa or other preferred hosts.

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**Figure 23-4.** Two-generation life history pattern for *Lygus hesperus* in a conifer nursery in southwest Oregon.

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## CHAPTER TWENTY-FOUR

# Root Weevils

## *Otiorhynchus sulcatus*; *O. ovatus*; *Sciopithes obscurus*

Joseph Capizzi

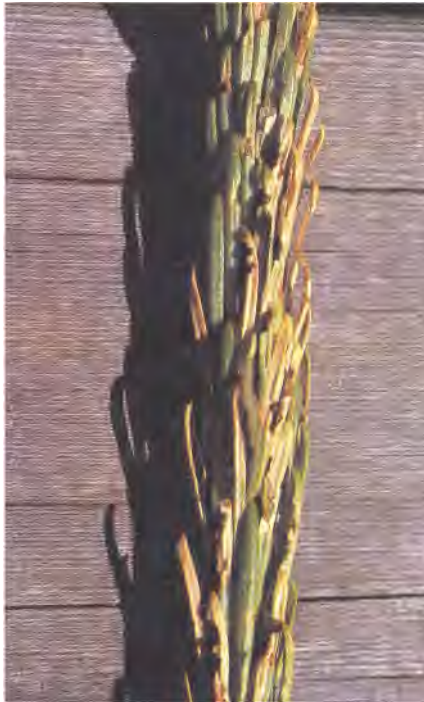
### Insects and hosts

Root weevils, primarily the black vine weevil, *Otiorhynchus sulcatus*; the strawberry root weevil, *O. ovatus*; and the obscure root weevil, *Sciopithes obscurus*, damage true firs and Douglas-fir in Northwest forest nurseries. These weevils have much in common: all females incapable of flight, they have a single generation of offspring per year, they lay up to several hundred eggs, they feed in darkness or subdued light, and they attack both roots (as larvae) and foliage (as adults) of conifer seedlings. Besides conifers, hosts in the Northwest are berries and rhododendrons.

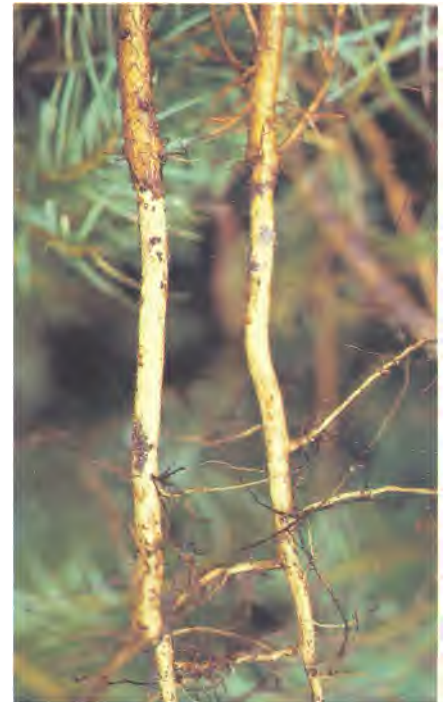
### Symptoms

Adult weevils notch the edges of the leaves of host plants (Figure 24-1). Signs of their feeding are very apparent in broadleaf plants but less so in conifers because the degree of injury per feeding incident is less. Often needles are severed and drop from the tree immediately, or are so damaged that they drop in a short time.

Adults feed mainly on new growth, resulting in sparse foliage. Larvae feed on small feeder roots first. As they grow, they feed on larger roots, often severely damaging or even killing plants by girdling their roots in the spring (Figure 24-2). Injured plants develop a greenish-yellow cast that does not respond to fertilization or watering.



**Figure 24-1.** The feeding of adult root weevils produced notches on the edges of these noble fir needles.



**Figure 24-2.** Damage from root weevil larvae on Douglas-fir. The cortical layer has been completely removed from most of the root system.

### Insect biology

These insects overwinter in the larval stage (Figure 24-3). Larvae are legless, C-shaped, and cream-colored with brown heads.

They quickly complete their development, pupate (Figure 24-4), and emerge as adults in the spring (Figure 24-5); or in the case of *Sciopithes obscurus*, in mid-summer.

Three to 4 weeks after adults emerge they begin to lay eggs. Eggs are laid singly or in small clumps. They are oval, sticky, and white at first but turn brown in a few days (Figure 24-6). They hatch in about 10

days. Young larvae then attack roots, feeding until the onset of cool weather in the fall. In *Otiorhynchus*, egg laying peaks in early July; in *Sciopithes obscurus* it peaks in August. The insects stop laying eggs in late September.

Some natural mortality of adults occurs throughout the summer following egg laying. Some adults overwinter in protected areas. The number of survivors depends on the severity of the winter weather and the shelter and moisture available. Adults that have overwintered are capable of laying eggs and do so in

the second spring of their lives. The impact of this overwintering adult population is not well understood.

## Loss potential

Root weevil larvae can be devastating to conifer seedlings. Adult damage is far less severe by comparison.

## Management

Since both larvae and adults usually go undetected until considerable damage is evident, a thorough knowledge of the insects and their habits is important. Also, because they are all female, their ability to multiply is great. The discovery of even a few weevils is cause for alarm.

### Root weevil larvae damage

appears:

**2+0, transplants**

**Summer to lifting**

### Root weevil adult damage

appears:

**1+0**

**Summer**

The search for emerging adults should begin in early spring when nights begin to get warm. The time of emergence must be determined in each nursery for best control. There are no sex pheromones—remember, these insects are all female. Deadfall traps are ineffective and time-consuming. Most effective is the use of an insect net and a head lamp. Sweep the insect net firmly across the conifer beds without damaging the seedlings. Weevils drop to the ground when disturbed and remain motionless for several seconds. This behavior and their color make them difficult to see and collect.



Figure 24-3. Root weevil larvae. Approximate size is 5 mm (1/4 inch) long.



Figure 24-4. Pupa of typical root weevil. Approximate size is 8 to 12 mm (5/16 to 1/2 inch) long.

Control measures should be initiated within 4 weeks of emergence, in order to kill weevils before they start to lay eggs.

### CULTURAL CONTROL

These pests are not in every forest nursery. Every effort should be made to keep them out if they are not present. Any movement of soil into noninfested areas should be restricted. These weevil species

cannot fly, but they can crawl. Keep native hosts away from nursery beds. Transplant beds should be cultivated and allowed to remain fallow between plantings. Mature larvae are quite susceptible to insect-eating nematodes, but the placement of the nematodes and the soil characteristics—particularly the moisture requirements for nematode survival—are critical.

**CHEMICAL CONTROL**

Adults—Chemicals may be used to reduce adult populations in the spring when the insects are feeding on new growth and before they have a chance to lay eggs. The chemicals will remain effective for several days. Even if weevils are not directly contacted, they feed at least every other day and thus will ingest treated foliage. Treatments should be repeated monthly throughout the egg-laying season. If damage is slight, adult populations are low, or sprays cannot be applied as often as indicated, most of the spraying should be done beginning 3 to 4 weeks after the first adults emerge and continuing through the peak emergence period.

Acephate, bendiocarb, parathion, and several synthetic pyrethroids have been found effective. Some trials indicate longer residual activity with synthetic pyrethroids. Parathion is a restricted-use insecticide that is extremely toxic. It may be used only by or under the super-

**Root weevil damage may be confused with:**  
**Fusarium root rot**  
**Grasshopper damage**  
**June beetle larvae damage**  
**Mechanical damage**

vision of a licensed applicator. Seedbeds may also be fumigated before sowing.

Larvae—Because larvae are hidden underground, control of them is more difficult than control of adults. Larvae may be controlled any time they are in the soil, but unfortunately, mature larvae and the pupal stage of the insect respond at best only slightly to chemical insecticides. In addition, the soil type may prevent penetration of chemicals, and organic matter in the soil may bind pesticides, reducing their effectiveness.



**Figure 24-5. Typical adult root weevils. Approximate size is 5 to 8 mm (1/4 to 5/16 inch) long.**



**Figure 24-6. Root weevil eggs. They are typically less than 1 mm (1/16 inch) in diameter.**

Spraying is recommended in summer, or fall, or both. Orthene and bendiocarb are recommended. The soil must be drenched through the root zone for the insecticide to be effective. Treating foliage and drenching soil results in control of both adults and larvae. Read and follow label directions for proper use.

Spraying for adult weevils is best done on warm nights when they are actively feeding. A high-pressure handgun offers the best application

method because it penetrates the foliage and provides good coverage. A boom with nozzles directed down over the beds, set at sufficient pressure and volume to wet them thoroughly, also offers effective application. Air applicators, mist blowers, and low-volume/low-pressure sprayers are less effective.

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## CHAPTER TWENTY-FIVE

# Minor Insects

David L. Overhulser

## Leafrollers

### Lepidoptera: Tortricidae

*Choristoneura rosaceana*; *Archips argyrospilus*

Both species of leafroller are widely distributed throughout the United States. Larvae feed on a broad range of deciduous trees, perennial plants, and herbaceous weeds. In the Willamette Valley, leafroller larvae also feed on the new growth of Douglas-fir seedlings

Leafroller damage appears:

2+0

Late spring through summer



Figure 25-1. Leafroller larvae measure 7 to 19 mm (1/4 to 3/4 inch) in length.

between June and August. The most obvious symptoms are the presence of silk webbing and small, pale-green to olive-green larvae 6 to 20 mm long (1/4 to 3/4 inch) on new shoots (Figure 25-1). Insecticides such as diazinon and acephate are effective against leafrollers.

## Seedcorn maggot

### Diptera: Anthomyiidae

*Hylema platura*

The seedcorn maggot is an important pest of vegetable crops throughout the United States. Twice it has been reported to damage germinating Douglas-fir seeds in western Oregon and Washington. In most crops, damage is correlated with late sowing during cold, wet springs when germination and growth are

slowed. Symptoms of an infestation are dying germinants and the presence of a yellowish-white maggot 3 to 7 mm (1/8 to 1/4 inch) long (Figure 25-2). Infested beds can be treated with diazinon or chlorpyrifos. However, like any pesticide treatment of germinants, this should be undertaken carefully because of the high potential for phytotoxicity.

Seedcorn maggot damage

appears:

1+0

Late spring through

early summer

## Lepidoptera:

### Pyraustidae

*Nomophila noctuella*

(no common name)

This insect is found throughout the continental United States and is a general feeder on young, succulent plants. There is a single report of its having caused cutworm-like damage to Douglas-fir seedlings. Larvae 13 to 22 mm (1/2 to 7/8 inch) in length clip seedlings off above the ground and carry the severed tops into a tunnel in the soil. A unique damage symptom is a silk thread leading from the tunnel to the base of the clipped seedling. Insecticides effective against cutworms, such as diazinon, should also control this insect.

## Cooley spruce gall adelgid

Homoptera: Adelgidae

*Adelges cooleyi*

The Cooley spruce gall adelgid is one of the most common insects found on Douglas-fir foliage in Oregon and Washington. The presence of the white, cottony tufts produced by the adult adelgid often causes alarm. However, most infestations do not significantly affect seedling vigor or survival. Damage appears as yellow spots on new foliage and distortion of elongating needles. Only the crawler stage of the insect is susceptible to control with insecticides. If control becomes necessary, carbaryl or endosulfan can be used against the crawlers as new growth flushes in the spring.

Cooley spruce gall adelgid  
damage appears:  
2+0  
transplants  
Spring through fall

## Migratory grasshopper

Orthoptera: Acrididae

*Melanoplus sanguinipes*

The migratory grasshopper can be an occasional pest on pine nursery stock during the summer months in eastern Oregon. The risk of seedling damage is greatest dur-

Grasshopper damage appears  
2+0  
Summer

ing the years when a grasshopper outbreak occurs. Adult grasshoppers 25 to 38 mm (1 to 1-1/2 inches) long are easily observed feeding on both new and old needles (Figure 25-3). Multiple



Figure 25-2. Seedcorn maggots and damaged Douglas-fir seedling. Mature maggots are about 6 mm (1/4 inch) long. Weyerhaeuser Company photo.

applications of insecticides such as diazinon or carbaryl may be necessary to protect seedlings from this highly mobile pest.

## Pine bark adelgid

Homoptera: Adelgidae

*Pineus* species

Bark adelgids can occur on pine seedlings grown in bareroot nurseries. These adelgids produce white, waxy secretions similar to those produced by Cooley spruce gall adelgid on Douglas-fir. Pine seedlings with heavily infested foliage or stems may become stunted and develop yellowish needles (Figure



Figure 25-3. Grasshopper feeding damage on ponderosa pine foliage in southern Oregon. Oregon Department of Forestry photo.



Figure 25-4. Pine bark adelgid secretions on lodgepole pine. Oregon Department of Forestry photo.

**Pine bark adelgid damage  
appears:  
2+0  
Summer**

25-4). The insecticide endosulfan can be used to control bark adelgid infestations.

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## CHAPTER TWENTY-SIX

# Nematodes

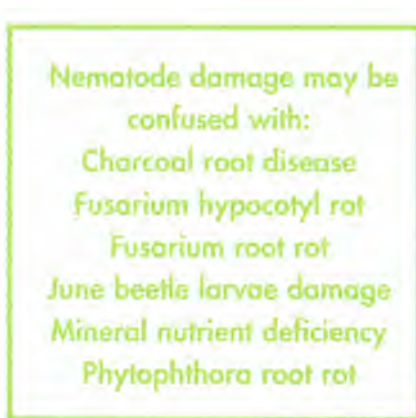
## *Pratylenchus penetrans*; *Xiphinema bakeri*

**Fred D. McElroy**

There are two nematodes of concern in forest nurseries, the root-lesion nematode, *Pratylenchus penetrans*, and the Baker dagger nematode, *Xiphinema bakeri*. While these nematodes produce similar above-ground symptoms, each has a unique biology and feeding habit and causes unique symptoms below the ground.

### Distribution and hosts

Both nematode species are indigenous to the Pacific Northwest. *Pratylenchus* is more widely distributed and has a broader host range. It attacks all species of conifers, but appears to be most damaging to Douglas-fir and true firs in the Pacific Northwest. *Xiphinema* has been observed to damage spruce (Sitka, white, and Engelmann), western



hemlock, Douglas-fir, and noble fir. The greatest damage occurs on Douglas-fir.

*Pratylenchus penetrans* has an extremely wide host range that



**Figure 26-1.** Field symptoms of *Pratylenchus* damage on Douglas-fir. Note chlorosis and differing degrees of stunting shown by the seedlings in the two beds.

includes many weed species. Nematode densities can be quite high on certain weed species, notably grasses, without a significant effect on the plant. On susceptible crops such as conifers, damage can be greater when weeds are present because nematode densities may be high under the weeds and because weed competition further weakens the conifers. The host range of *X. bakeri* is more limited. This species does not interact with weeds as *P. penetrans* does.

### Symptoms

The above-ground symptoms of damage from both *Pratylenchus* and *Xiphinema* resemble those caused by any root-destroying organism.

Infected seedlings may be stunted, chlorotic, or unthrifty; or in severe cases may even die. Symptoms may be confused with those of mineral deficiency, since root damage by these nematodes may reduce mineral uptake even when soil fertility is adequate. Infected plants are more susceptible to drought stress during periods of low soil moisture than are uninfected plants.

The distribution pattern of nematode damage in the nursery is characterized by circular or irregular patches of stunted seedlings. These patches cut across rows and beds. Stunting is observed in a gradual progression from noninfested areas to infested areas (Figure 26-1). In contrast, other types of soilborne problems are frequently characterized by a marked difference in size



between contiguous healthy and affected seedlings.

Each nematode species causes unique damage below the ground. *Pratylenchus* causes necrosis of the cortex, resulting in general decay of the feeder roots (Figure 26-2). During the early stages of infestation the feeder root terminal is destroyed, stimulating proliferation of lateral roots above that point. This process is repeated many times on the newly developing roots, resulting in a proliferation of the root system known as witches' brooming. This type of root damage is a good early clue to severe infestations of *Pratylenchus*.

Damage from *P. penetrans* differs from that caused by *Phytophthora*, which exhibits a similar symptom, in that the necrosis is not as obvious and there is no sharp line between living and dead tissue. However, with increased destruction by the nematode, all feeder roots are killed and the seedling eventually dies from an inability to take up nutrients and moisture. Frequently fungi enter the stressed tissue and hasten mortality.

Roots of seedlings attacked by *Xiphinema* are dark, swollen, and often club-tipped, and have few if any laterals. A condition known as corky root disease frequently develops as the result of infection by the fungus *Cylindrocarpon destructans* following feeding by the nematode. This corky, swollen, and deformed root tip can often be used to diagnose *Xiphinema* in the field. However, other conditions can cause a similar appearance. A nematode assay of the soil should be made for confirmation.

## Nematode biology

Both *Pratylenchus* and *Xiphinema* are microscopic, thin, colorless roundworms. They have a hypodermic-needle-like feeding structure called a stylet, which is used to puncture plant cells and deliver digestive gland fluid into them (Figure 26-3). This fluid predigests



**Figure 26-2. A healthy Douglas-fir seedling and two damaged by *Pratylenchus*. Note the stunting and lack of root development on the two damaged seedlings.**

the contents of the cell, which are then drawn into the nematode through the stylet by a muscular pumping bulb.

*Pratylenchus* is much smaller (0.5 mm) than *Xiphinema* (2 mm), and has a shorter stylet. After feeding on a cell for a period of time, *Pratylenchus* cuts a hole in the cell wall and enters the cell. As it repeats this procedure, cell after cell, the nematode penetrates deep into the cortex of the root. Intensive feeding in a limited area of the root frequently causes a lesion, from which the nematode derives its common name.

By contrast, *Xiphinema* possesses a very long stylet, which is the only part of the nematode that penetrates the root tissue. Feeding activity causes the tissues to swell and prevents the formation of lateral feeder roots, but roots do not rot.

As the *Pratylenchus* female moves through the soil and roots, she deposits one or two eggs per day along the way. The first-stage

larva develops within the egg and goes through one molt to become a second-stage larva, which emerges in about a week. The larvae begin to feed on the root. They go through three more molts before becoming adults, a process that takes about 50 days (Figure 26-4). The life cycle of *Xiphinema* is similar, but reproduction is slower; the cycle takes almost 2 years to complete.

While all stages of the nematode are active throughout the year in the Pacific Northwest, the rate at which these processes occur increases during the warmer summer months. Population densities are generally tied to root growth activity—the nematode population increases after a period of plant growth.

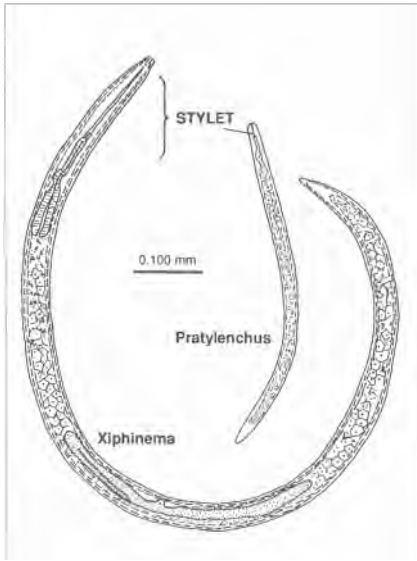
Soil types most suitable for conifer nurseries are also ideal for the proliferation of both *Pratylenchus* and *Xiphinema*. Populations can build to high numbers in loamy soils. Heavier clay soils, because of their higher moisture-retention capacity and lower oxygen supply, tend to restrict growth of the nematode population.

**Nematode damage appears:**  
 1+0, 2+0  
 Mid-summer (1+0)  
 through fall (2+0)

*Xiphinema* is best adapted to sandy soils. These soils tend to have a low nutrient-holding capacity and, in combination with nematode damage and the fungus *Cylindrocarpon*, are conducive to the development of corky root disease.

## Loss potential

Loss can occur in many forms. At worst, nematodes may kill seedlings. At best, they may act



**Figure 26-3. Size comparison of *Xiphinema* and *Pratylenchus*, showing size and location of stylets.**

only as contaminants of roots, from which they may be introduced into a new environment. In between these extremes, seedlings may survive but may not make packing standards; severely infected seedlings must usually be culled at the time of lifting. Damaged seedlings may be inefficient users of water and nutrients. They may have difficulty surviving or may remain stunted when transplanted to a new environment. These seedlings are more susceptible to damage from other organisms and from environmental factors such as frost and heat. Frost heaving often occurs on seedlings with poorly developed root systems.

The younger the seedling or the smaller its root system, the more susceptible it is to damage from nematodes. *Pratylenchus* populations of fewer than 500 per pint of soil may cause severe stunting or even mortality of a 1-year-old seedling, but only moderate stunting of a 2-year-old or 1+1 seedling. By the time the seedling is at the 2+1 stage, much higher densities of nematodes—greater than 1,000 per pint of soil—are necessary for significant damage. Similar damage may result from lower densities of

*Xiphinema*, however. The degree of damage caused by *Pratylenchus* to 1+0 and 2+1 seedlings as described above requires *Xiphinema* populations of only about 100 and 500 per pint of soil, respectively.

One-year-old seedlings, either bareroot or container-grown, are frequently damaged severely when transplanted into nematode-infested soil. These stock types have the disadvantage of a limited root system, have suffered pruning and transplant shock, and frequently lack significant mycorrhizal development, and then are introduced into an environment where nematodes are already established. Nematodes immediately attack the newly developing roots and prevent the seedlings from becoming well established.

Both nematode species interact with soil fungi on a wide range of crops. Feeding by *Pratylenchus* not only creates wounds on the roots through which other organisms can enter, but also alters the host's physiology, breaking down resistance to soil fungi. It has not been determined whether this phenomenon occurs in conifers, but it has been well demonstrated in other crops.

Mycorrhizae apparently play an important role in protecting seedlings from nematode damage. Seedlings lacking mycorrhizae suffer more damage from nematodes, and the presence of nematodes prevents mycorrhizal development. The direct damage caused by the nematode and the prevention of mycorrhizal establishment both contribute to unthrifty growth and stunting of the seedlings.

## Management

Nurseries located on old forest sites rarely encounter a significant problem from nematodes. Nematodes on such sites are usually few and spotty in distribution and are consequently easy to control. Old agricultural sites, however, have a wider distribution of nematodes at

higher, frequently damaging densities. Such sites may also harbor nematodes deeper in the soil profile, making control more difficult than for forest sites.

Routine fumigation of seedbeds prevents nematodes from becoming a problem during the first 2 years of seedling growth. If seedbeds are not routinely fumigated, assays for nematodes should be done because the potential for damage is much greater at the early stage of seedling development than later. Damage is also frequently encountered in unfumigated transplant blocks within the nursery.

Each block within the nursery should be surveyed for the presence of nematodes. If no nematodes are detected and proper sanitation procedures are maintained, there will probably be no need for further assays. Nurseries that routinely grow transplant stock from other bareroot facilities should obtain a laboratory report from the source nursery for each lot certifying that the incoming stock is free of nematodes. A periodic assay of the transplant blocks is a further safeguard.

## CULTURAL

*Xiphinema* is easier to control than *Pratylenchus*, since it remains generally in the upper 10 centimeters of the soil and does not enter into the roots. *Xiphinema* has been controlled by fallowing in combination with disking or rototilling of infested soils during the hot, dry days of August and September.

## CHEMICAL

Many fumigants are available for controlling nematodes before planting. A combination of methyl bromide and chloropicrin controls nematodes, soil fungi, insects, and weed seeds. It is the most costly chemical treatment. Dazomet and metam sodium also control the same range of organisms to varying degrees. They are safer to handle, and metam sodium is slightly less expensive than the methyl bromide-chloropicrin combination. Dichlo-

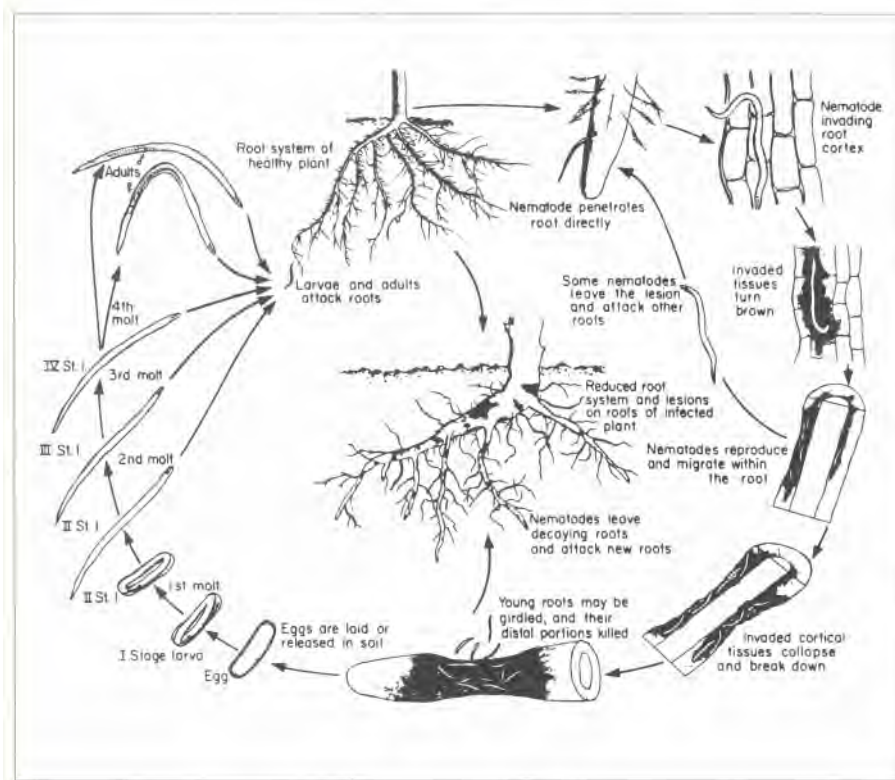


Figure 26-4. Life and disease cycle of *Pratylenchus*. Reprinted from *Plant Pathology*. Used by permission.

ropropene is primarily a nematicide, but it also affords control of soil fungi when combined with varying amounts of chloropicrin. It is the least costly of the soil fumigants.

Postplant nematicides such as fenamiphos and oxamyl have been tested and found effective. They belong to a class of compounds called contact nematicides, which prevent nematode feeding and reproduction.

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## CHAPTER TWENTY-SEVEN

# Cold Injury

## Frost Damage; Frost Heaving; Winter Desiccation

**Richard K. Hermann**

Cold injury is the result of plant cells killed by temperatures at or below freezing. The extent and degree of injury depend on many factors, and predicting exactly when frost damage is going to occur is difficult.

**Frost damage may be confused with:**

**Mechanical damage**

**Pesticide damage**

**Phomopsis canker**

**Sirococcus tip blight**

**Tip blight of pine**

**Upper stem canker**

**Symptoms appear:**

**1+0, 2+0, transplants**

**Late spring and early fall**

A good description of how frost can damage plants is given by McDonald, who says that frosts can be divided into two categories: radiation frosts, and wind and advection frosts. The two types may occur simultaneously, and in some instances a radiation frost may intensify a wind frost.

Radiation frosts occur on cloudless nights with little or no wind, when excessive amounts of heat energy in the soil and plants are lost to the sky as long-wave radiation. Temperatures of leaves, air near the ground, and the soil surface may fall below freezing. Advection frosts occur when cold air flows from a

higher location to a lower one, displacing lighter, less dense warm air. Such flows will pool and concentrate if they encounter an obstruction, causing a "frost pocket." Wind in excess of 4 mph from cold regions causes a wind frost, which can occur at any time of the day or night and is not necessarily related to topography.

Two other types of cold-weather damage occur in Pacific Northwest bareroot nurseries: frost heaving and winter desiccation. Frost heaving leaves seedling roots partly or totally uncovered due to alternate freezing and thawing of soils. Winter desiccation is the result of cold or frozen soils inhibiting uptake of moisture by roots so that part or all of the seedling top desiccates and dies.

### Occurrence

Woody plants of the temperate zones undergo a distinct seasonal change in their ability to withstand cold. As a general rule, they are damaged easily by temperatures of 0 degrees C (32 degrees F) or just below during the growing season, but can stand much lower temperatures without damage during dormancy. The hardening and dehardening of plants do not occur on the same dates each year. Frost resistance is influenced by a combination of environmental factors, primarily temperature, day length, and moisture—factors that vary from one year to the next.

Seedlings are most often injured in fall when they have not acquired enough coldhardiness to withstand



**Figure 27-1.** Cold-damaged Douglas-fir seedling; typical needle yellowing later turns brown. Oregon Department of Forestry photo.

early frosts, and in spring when a late frost occurs after seedlings dehardened. Frost damage can occur in all Pacific Northwest nurseries, although the degree of potential damage varies with the location of the nursery.

Susceptibility of seedlings to frost damage varies among forest tree species commonly grown in nurseries. Frost resistance is high in lodgepole pine, ponderosa pine, incense cedar, Engelmann spruce, and true firs; medium in the coastal variety of Douglas-fir and in western hemlock and western redcedar; and low in Sitka spruce.

However, sensitivity to frost is by no means uniform within a species,

particularly in those with a wide geographical distribution. Even seedlings from the same seed source may vary widely in their susceptibility to frost. As a general rule, seed sources from high elevations and higher latitudes are more frost-resistant than those from low elevations and lower latitudes. The date of bud set can be used as general indicator of coldhardiness. Provenances that set winter buds early tend to suffer less from fall and winter frost than those that set winter buds late in the season.

Frost resistance varies also among parts of a seedling. Leaves are generally the most hardy and roots the least hardy organs, with stems and buds occupying an intermediate position. Frost resistance may vary even within the same plant organ. In a stem, cells of the phloem and cambium are

**Winter desiccation may be confused with:**  
**Lower stem canker**  
**Pesticide damage**  
**Phoma blight**

**Symptoms appear:**  
**2+0, transplants**  
**Spring**

more sensitive to frost than cells of the cortex or the xylem.

To complicate matters further, the annual cycle of hardening and dehardening is not synchronized for all parts of a seedling. In some species, needles reach their peak of frost resistance 4 to 6 weeks later than stems, and 2 to 4 weeks later than buds.

*Needles*—Frost injury to needles changes their color to a reddish-brown, sometimes preceded by a purplish or dull gray hue (Figures 27-1, 27-2, and 27-3). Newly killed needles, however, cannot be distin-



**Figure 27-2. Large-scale damage to Douglas-fir seedlings because of freezing temperatures. The seedlot in the foreground was more coldhardy and is apparently unaffected.**



**Figure 27-3. Frost damage to recently flushed foliage of Douglas-fir. Oregon Department of Forestry photo.**

guished visually from uninjured needles because this color change takes days—sometimes weeks or even months—to occur. In general, the color of the needles will change sooner if seedlings were injured in spring rather than in fall or winter when temperatures tend to remain low.

It is possible to check for damage immediately after a cold spell, however. Electrical impedance of cell membranes is altered when membranes are damaged. Impedance can be measured with special equipment, and damaged tissue can be thus detected as soon as it has thawed out.



**Figure 27-4.** A slice through the stem of a Douglas-fir seedling shows the browning of the cambium caused by freezing damage.

*Stems*—*Injury* to stem tissues can be observed by slicing away strips of bark. Injured stems show various degrees of browning (Figure 27-4). Rarely, stems will show visually recognizable external signs such as shriveled or deep-reddish-brown bark. If the phloem has been killed, bark will feel mushy and can be peeled from the stem easily. If frost occurred without snow cover on nursery beds, the first place to look for freezing injury is close to the ground because that is where temperatures are lowest and thus where frost injury is likely to occur first. If seedlings were covered partially by snow, stems should be examined just above the snow line.

Another indication of damage to the stem is shedding of needles. Needles may be shed even if they themselves were not killed by frost; this appears to be associated with injury to the needle traces and cortical tissues of the stem. If both needle and stem tissue are damaged, needles will begin to drop after 2 to 3 weeks. If only needles are damaged, they tend to remain

attached to seedlings for several weeks or even months before they drop.

Winter desiccation can affect both needles and stems. This injury occurs when a low-humidity wind blows across the seedlings at the same time that cold wintertime temperatures freeze the upper soil layer, preventing or restricting moisture uptake by the roots. The needles can literally dry up. Often a snow cover prevents damage by protecting the seedlings from the wind or by preventing the ground from freezing, or both. Still, damage will occur to any portion of the seedling above the snow layer.

*Buds*—*Cold* injury to buds is not immediately apparent externally. Four to 6 weeks after they are frozen, injured buds begin to take on a dried-up appearance. However, buds can be checked for injury a few days after exposure to freezing temperatures by cutting them open (Figure 27-5). Tissue inside a frost-damaged bud will show light brown to dark brown discoloration—the darker the tissue, the more severe the injury.

*Roots*—Roots probably sustain less damage from frost than do the

above-ground parts of seedlings because the soil temperature falls much more slowly than the tem-

**Frost heaving may be confused with:**  
**Cutworm damage**  
**Damping-off**  
**Seedcorn maggot damage**

**Symptoms appear:**  
**1+0**  
**Winter through spring**

perature above ground. Soil often remains unfrozen even when above-ground temperatures fall below freezing. Moreover, the limited information available indicates that frost resistance ratings for above-ground parts of seedlings do not necessarily apply to their roots. In addition, tolerance to root damage from frost varies with species. Roots of western hemlock have been shown to be hardier than roots



**Figure 27-5.** Cold-injured and noninjured lateral bud. Note that the center area of the left lateral bud is brown, in contrast to the green center of the bud on the right.



**Figure 27-6.** Frost heaving in small 1+0 seedlings.

of ponderosa pine and noble fir.

Injured roots show brown or almost black discoloration when bark is stripped away. Injured bark becomes mushy and can simply be pulled off. At this stage, however, freezing injury can easily be mistaken for symptoms of fungal diseases. Caution is necessary to avoid a wrong diagnosis.

Freezing and thawing of the upper soil surface are responsible for frost heaving. This condition, while not actually an injury, leaves seedling roots partially or totally uncovered (Figure 27-6). The expansion and contraction slowly pulls seedlings out of the ground. Larger seedlings with well-developed root systems generally are not affected, though roots can take on a corkscrew appearance in severe cases of repeated freezing and thawing of the soil surface (Figure 27-7).

### Predisposing factors

Irrigation schedules that extend active growth of shoots well into the fall predispose seedlings to damage from early frosts. Insufficient irrigation, particularly during the second or third year, limits the accumula-

tion by seedlings of carbohydrate reserves. This contributes to lowered frost resistance and may result in freezing injury during dormancy.

Nutrition can influence the seasonal growth pattern of seedlings and thus alter their susceptibility to freezing injury. Fertilization may result in prolonged growth in fall or earlier bud burst in spring, thus increasing the risk of frost damage. Nutrient stress also reduces frost hardiness.

### Loss potential

The potential for loss is considerable but difficult to quantify because of the many variables involved. Seedlings will die if the roots or the cambium and phloem of the lower part of the stem have been killed. Injury to needles is less serious; it will weaken the seedling but not necessarily kill it. Douglas-fir may recover if uninjured buds are left on the stem.

### Management

Overhead irrigation is an effective means of protecting seedlings from radiation or advection frosts. Water droplets suspended in the air reduce heat loss by lowering the flow of outgoing long-wave radiation. Irrigation prevents frost by increasing the thermal conductivity and heat capacity of the ground and by making possible the release of latent heat when the water freezes (Figure 27-8). The temperature of seedlings will not fall below the freezing point as long as the freezing process—the changing of water from the liquid to the solid phase—continues to take place. Overhead sprinkling, however, does not protect against freezing injury when temperatures drop below  $-7$  degrees C (20 degrees F). Sprinkler systems usually cannot be kept in operation at such low temperatures anyway.

If applied improperly, however,

overhead irrigation may be ineffective or may even increase frost damage. Seedlings sprinkled with insufficient water during a light radiation frost may suffer serious damage. This may be because when the air is dry, the temperature of the wetted needles will approach the wet-bulb temperature, which may be significantly lower than the dry-bulb temperature. Another possible reason is that the small amount of ice that forms on a leaf will prevent the undercooling of the cell solution and also may dilute the solution, thereby raising the freezing temperature.

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**Figure 27-7.** Corkscrew-appearing root system due to repeated soil freezing (expansion) and thawing (contraction).

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**Figure 27-8. Frost control in a bareroot nursery. The release of latent heat as the water freezes prevents the temperature of the seedling from falling below 0 degrees C ( 32 degrees F ). Once the air temperature drops below -7 degrees C ( 20 degrees F ), injury can no longer be prevented by this method. Photo courtesy of IFA Nurseries, Inc.**



## CHAPTER TWENTY-EIGHT

# Heat Injury

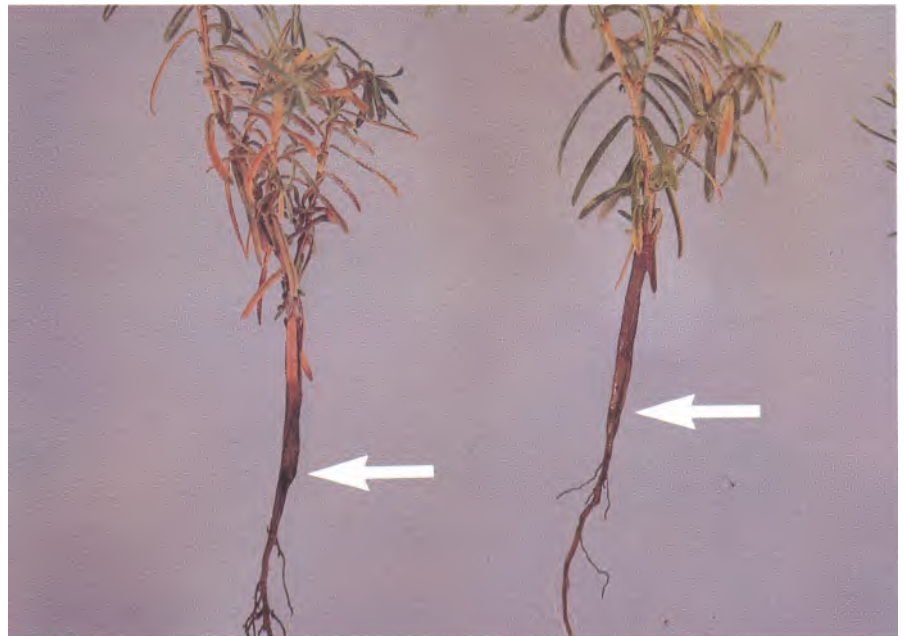
**Richard K. Hermann**

The surfaces of nursery beds in the spring and summer can reach temperatures lethal to seedlings unless measures are taken to prevent the buildup of heat. There are two general types of heat injury; seedlings may suffer from one or both. Injury from direct heat causes the cells of the plant to collapse and the plant to topple over and die. Indirect injury causes disturbances in the plant's metabolic processes, such as the denaturation of proteins. Its symptoms are less immediately obvious and more difficult to identify than those of direct heat injury. Effects of indirect injury are widely variable; plants may suffer only minor reversible damage, or they may die. No statistics are available on the extent of seedling losses from heat injury.

**Heat damage may be confused with:**  
**Cutworm damage**  
**Damping-off**  
**Frost heaving**  
**Fusarium hypocotyl rot**  
**Seedcorn maggot damage**

## Occurrence

Injury usually occurs on the stem just above the ground, where the buildup of heat is greatest. Sometimes heat injury occurs on cotyledons just as they emerge from the soil. Heat injury in older



**Figure 28-1.** Heat damage to stems of 1+0 seedlings at the groundline. Damage to phloem tissue prevents photosynthates from reaching roots, causing an enlargement of stem tissue above the damaged area.

seedlings is rare, both because the lignification of their stems provides some protection, and because the development of shoots causes the canopy to close on seedbeds, significantly reducing the buildup of heat on the ground. Numerous studies indicate that injury is likely to occur at temperatures above 49 degrees C (120 degrees F). Temperatures in excess of 60 degrees C (140 degrees F) will quickly lead to death.

## Symptoms

Direct heat injury in a young seedling (4 to 6 weeks old) first becomes apparent when the basal

part of the stem takes on a water-soaked appearance. This is caused by the rupture of cortical cells. With continued exposure of the seedling to heat, a lesion forms on the lower stem. It looks like a white streak or spot, and has given rise to the name "whitespot disease" for heat damage. If temperatures remain high, the lesion expands until it encircles the stem, constricts it, and finally causes the seedling to topple over.

In seedlings older than 4 to 6 weeks, heat-damaged areas are dark and sunken. Seedlings of that age have usually produced enough secondary xylem to hold them erect unless the constriction is too deep. Such seedlings can sometimes live

for several months after heat damage because even a deep constriction does not stop upward movement of water to the leaves. However, since swellings above the constriction impede phloem transport of photosynthates to roots, seedlings eventually die (Figure 28-1).

## Management

All coniferous species grown in nurseries in the Pacific Northwest are susceptible to heat damage as young seedlings. The principal method for protecting seedlings against heat injury is to irrigate them so that the evaporating water

**Heat damage symptoms  
appear:  
1+0  
Late spring through summer**

cools the seedbeds (Figure 28-2). Besides cooling the soil surface, sprinkler irrigation during the hottest part of the day can lower the air temperature 10 to 15 degrees F or more, cooling the foliage of seedlings and reducing their overall heat stress. However, the frequency and length of irrigation needed for effective protection varies with temperature, seedling development, soil type, and tree species. Seedling nurseries should develop their own guidelines for cooling seedlings with irrigation.

Shading is an effective method of protecting seedlings from heat injury. However, materials for shade frames are expensive, and installing and removing them is labor-intensive.



**Figure 28-2.** Losses from heat can be minimized by shading seedlings or cooling them with water. Photo courtesy of Alan Kanaskie.

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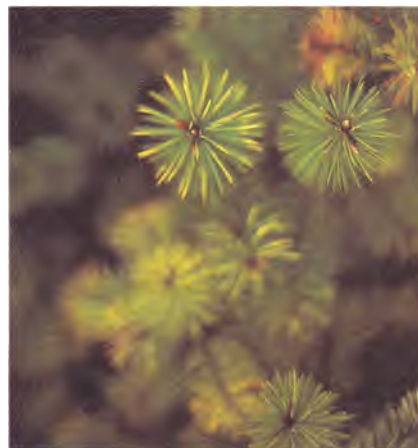
## CHAPTER TWENTY-NINE

# Mineral Nutrient Problems

**Thomas D. Landis**

Mineral nutrients are taken up by the plant from the soil solution in various ionic forms. These ions are replenished through mineral decomposition and decay of organic matter and from fertilizer amendments. Mineral nutrients have a significant effect on plant growth rate. A mineral nutrient deficiency exists when the plant's growth rate is limited by the availability of a certain nutrient. If plants are supplied with an excess of a certain mineral nutrient, however, they may continue to take it up until growth is retarded. This is referred to as a mineral nutrient toxicity. In addition to deficiencies and toxicities, mineral nutrient disorders can also be caused by an imbalance in the relative availability of different nutrients.

The 13 essential mineral nutrients are customarily classified into two categories: macronutrients and micronutrients. Macronutrients, which are used by plants in relatively large amounts, include nitrogen, phosphorus, potassium, calcium, magnesium, and sulfur. The seven micronutrients, iron, manganese, zinc, copper, boron, molybdenum, and chloride, are required in very small amounts, and the difference between deficiency and toxicity can be quite small.



B



C

**Figure 29-1.** Chlorosis can be caused by a deficiency of a number of mineral nutrients. For example: (A) nitrogen, (B) magnesium, (C) iron.

## Occurrence: species and season

All seedling species and stock types are susceptible to both deficiencies and toxicities of mineral nutrients, although different species may express these disorders with different symptoms. Young and actively growing seedlings are most susceptible to nutritional problems.

## Symptoms

### DEFICIENCIES

When a seedling is unable to obtain enough of a mineral nutrient, the first effect is a reduction in growth rate. Often this initial growth reduction goes unnoticed because this "hidden hunger" is not accompanied by visible symptoms. Eventually, however, nutrient-deficient seedlings may become stunted.

If the mineral nutrient deficiency is not corrected, the seedling may exhibit certain visible deficiency symptoms. These can be useful in diagnosing the deficiency and prescribing corrective fertilizer amendments. Some common deficiency symptoms, such as nitrogen chlorosis, are well known to experienced nursery managers.

Unfortunately, however, different species of seedlings exhibit mineral nutrient deficiencies in different ways. Phosphorus deficiency, for example, is expressed as foliar discoloration that varies with species, ranging from dull green to purple in color.

To complicate matters, some symptoms can be caused by a deficiency of any of several different mineral nutrients. For example, chlorosis (yellowing) can be caused by a deficiency of nitrogen, magnesium, or iron (Figure 29-1). Sometimes mineral nutrient deficiencies can be identified in the cover crop—phosphorus deficiency in oats, for example (Figure 29-2). Deficiency symptoms alone are not particularly helpful when dealing



**Figure 29-2.** Mineral nutrient problems can also be recognized in the cover crop. This chlorosis in oats is caused by a deficiency of phosphorus.

with the problem of multiple deficiencies; these obviously can become quite complicated. Nevertheless, deficiency symptoms can help identify nutritional problems when considered in conjunction with soil and foliar nutrient tests and practical experience. Some nursery manuals contain lists of deficiency symptoms for tree seedlings, but new seedling growers should seek the advice of experienced nursery managers or specialists.

### TOXICITIES AND IMBALANCES

Mineral nutrient toxicities and imbalances are particularly difficult to diagnose because an excess of one nutrient may induce a deficiency of another. Extreme toxicities are expressed as needle tipburn or leaf margin scorch, the typical symptoms of fertilizer burn (see Figures 31-1B and 31-1C). Foliar symptoms have been useful in diagnosing toxicities of certain micronutrients, such as manganese (Figure 29-3), but these diagnoses should be made in conjunction with nutritional analysis of seedling tissue. Experienced nursery

managers or specialists should also be consulted.

## Predisposing factors

Seedling nutritional disorders can occur without any predisposing stress factors, although some environmental conditions may indirectly inhibit the uptake of nutrients. Soil factors, especially pH, can have a significant effect on mineral nutrient availability. Because most conifers grow best in slightly acid soils, nursery managers try to maintain soil pH in the range between 5.0 and 6.0. Hardwood species can tolerate a slightly higher pH, around 6.0, although the requirements of individual species vary.

Any injury or disease that weakens or destroys the fine feeder roots can lead to nutrient deficiencies. The chlorosis that is symptomatic of some root diseases, such as *Phytophthora* root rot, may actually be caused by a mineral nutrient deficiency. Heavy rainfall or over-irrigation can lead to waterlogging of the soil, resulting

in anaerobic conditions in the root zone. When roots are unable to respire normally, mineral nutrient uptake is altered. Prolonged water stress can also cause mineral nutrient deficiencies, because many nutrient ions are absorbed into the root system with normal transpiration water uptake.

## Loss potential

Mineral nutrient deficiencies rarely result in seedling mortality. Growth losses do occur, however, but are impossible to quantify because they typically begin before foliar deficiency symptoms become visible. Stunted seedlings or those with visible nutrient deficiency symptoms are normally culled out on the grading table, but seedlings that are merely weakened by nutritional problems may not be identifiable. Nutritionally weakened seedlings are at a disadvantage on the outplanting site, and suffer severe transplant shock and slower initial growth.

**Mineral nutrient problems  
appear:  
All ages  
Any time throughout rotation**

## Management

Mineral nutrient deficiencies are easily managed and should not be a serious problem in a modern forest nursery. They can be prevented by regular testing of soils and seedlings, proper soil management, and well-planned applications of the proper fertilizers.

### NUTRIENT ANALYSIS

Soils from areas designated for seedling production should be sampled and tested for mineral nutrient content the fall before sowing, so that presowing applications of fertilizer can be incorpo-



**Figure 29-3. Foliar symptoms can be used to identify toxic levels of certain nutrients. This is manganese damage in spruce. Such diagnoses should be made in conjunction with tissue analysis.**

rated into the soil. This is especially important for nutrients like phosphorus and calcium, which are not mobile in the soil and therefore cannot be applied as a top dressing over the seedling crop. Soil pH can also be adjusted at this time by applying limestone or dolomite to raise pH or sulfur to lower it.

Both seedlings and soil should be analyzed for nutrients at the end of the first growing season, so that fertilizer programs may be adjusted during the second year. To ensure the accuracy of the analysis, soil and seedling samples should be collected systematically. Considerable variation can occur within a nursery, so sampling schemes must be designed to reflect this variability. Contact the testing laboratory before collecting samples. Laboratory personnel can often recommend a scientific sampling technique and give advice on handling and shipping of samples.

Many laboratories offer soil and seedling testing services. Because

tree seedlings have different nutritional requirements than agronomic crops, nursery managers should patronize laboratories that have experience with tree-nursery crops. Each laboratory uses slightly different analytical techniques; this may affect the test results. Nurseries should continue to patronize one laboratory so that results are comparable from year to year. Interpretation of test results requires experience. Laboratories that have previously tested tree seedlings and have accumulated test results from other seedling nurseries will be able to provide better recommendations than inexperienced laboratories. Nursery manuals contain soil fertility targets for tree seedlings, but these values must be interpreted with an understanding of the soil conditions and species at a particular nursery.

### SOIL MANAGEMENT

Many different soil factors affect seedling nutrition. Nursery managers can improve the nutritional status of their crop by using proper soil-management practices. The sandy soils that are best for tree seedling production are inherently infertile; growers need to maintain soil fertility with organic amendments and cover or green-manure crops. Organic matter has both physical and chemical effects on the soil: it increases the cation exchange capacity, and it improves the physical structure of the soil. Cover or green-manure crops also function as "catch crops" that supply readily available forms of mineral nutrients as they decompose. Legumes fix atmospheric nitrogen which is subsequently released to the seedling crop. Beneficial soil microorganisms, notably mycorrhizal fungi, have been shown to increase the availability of certain mineral nutrients, specifically phosphorus. Nursery managers should use cultural practices that encourage mycorrhizae.

**FERTILIZER APPLICATIONS**

The most practical and effective way to control soil fertility in forest nurseries is with fertilizers. Fertilizer application rates should be prescribed using previous experience or general recommendations, but the best practice is to base prescriptions on soil and seedling test results, taking into account experience with the particular species of seedling. Fertilizer application equipment should be calibrated carefully and tested regularly to make sure it is functioning properly (Figure 29-4).

The timing of macronutrient fertilizer applications should be based on seedling phenology, rather than calendar date, and on the characteristics of individual fertilizers. Certain immobile nutrients like phosphorus and calcium should be incorporated before sowing. Phosphorus fertilizers are most effective when banded alongside or under the seed. Nitrogen and potassium are more mobile and therefore may be applied as top dressings during the growing season.



**Figure 29-4.** Differences in foliage color indicate uneven application of fertilizer from improperly calibrated equipment.

Newly sown seedbeds are usually not fertilized until several weeks after seedlings emerge. Fertilizers can burn young, succulent seedlings, and high levels of nitrogen are thought to stimulate soil pathogens. Excessive nitrogen fertilization will also promote shoot growth at the expense of root growth, resulting in a seedling with a poor shoot-to-root ratio. With older seedlings or transplants, growers should not wait until deficiency symptoms appear, but should apply fertilizers based on periodic nutritional analyses of the soil and of seedling tissue. Later in the growing season, high nitrogen fertilization rates may delay hardening of seedlings.

Most nursery soils are not normally deficient in micronutrients, but proven deficiencies can best be treated with foliar applications of chelate fertilizers. Because of the narrow range between deficiency and toxicity of most micronutrients, individual-element fertilizers should be used rather than commercial micronutrient mixes.

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## CHAPTER THIRTY

# Pesticide Phytotoxicity

**Thomas D. Landis**

Nursery managers have traditionally used a number of chemical pesticides to help manage pest populations in forest nurseries. Nursery pesticides can be separated into two classes: nonselective pesticides, which are toxic to all organisms, and selective pesticides, which are applied to control a specific pest organism but are not intended to injure nontarget organisms (the crop seedlings or beneficial organisms).

Most nonselective pesticides are used only in areas that do not currently contain seedlings. Many nurseries use soil fumigants to reduce fungal and weed seed populations before sowing. Nonselective herbicides are often used to control weeds that develop in noncrop areas such as irrigation lines and road ditches. Some nurseries apply nonselective pesticides



**Figure 30-1.** Pesticide damage on a white pine progeny trial. Susceptibility was related to seed source.

Pesticide phytotoxicity  
may be confused with:  
Frost damage  
Lygus bug damage  
Mineral nutrient problems  
Salt injury

between rows of seedlings, using specially designed sprayers that shield the crop trees.

Selective pesticides can be applied directly to the seedling crop. Fungicides and insecticides are selective pesticides that rarely injure the crop seedlings unless they are improperly applied. A

relatively few selective herbicides are currently being used in forest nurseries. All nursery pesticides have been carefully screened for phytotoxicity before they are registered for use on tree seedlings, but pesticide injury still occurs. Most phytotoxicity incidents can be attributed to improper application of selective pesticides, drift of nonselective pesticides, or residual pesticides in soil.

**Improper application of selective pesticides.** Instructions for the proper use of a pesticide are listed on its label. Pesticide chemicals should be applied only by trained personnel; some registered pesticides can be purchased only by certified applicators. In spite of these familiar instructions,

cases of phytotoxicity can usually be traced to some type of applicator error. Failure to completely read and follow label instructions and failure to correctly calculate pesticide dilutions or application rates are common mistakes. Pesticide equipment must be properly calibrated, regularly checked, and cleaned between applications. Applying pesticides during unfavorable weather conditions, such as hot or windy weather, can also lead to phytotoxicity problems.

**Drift of nonselective pesticides.** Fumigants and other nonselective chemicals may drift from treated areas and damage adjacent crop seedlings. The amount of drift depends on the

characteristics of the pesticide, the application method, and local weather conditions. Because all fumigants convert to a gas in the soil, the treated area is usually covered with a plastic tarp or the soil surface is sealed with water. During temperature inversions, fumigants can drift and become concentrated in low areas of the nursery. Even liquid sprays can drift if a volatile ester formulation is used or if the pesticide is applied with a fine spray nozzle.

#### **Residual pesticides in soil.**

Soil sterilants are designed to provide an extended period of weed control and therefore can persist in the soil for many years. Because of this residual action, sterilants can cause phytotoxicity when treated land is subsequently converted to a seedling crop. Some herbicides that are safe for one crop may persist in the soil and harm subsequent crops. Fumigants applied to cold, wet soil may also persist longer than usual and cause phytotoxicity after the normal waiting period.

### **Occurrence: species and season**

All seedling species and stock types are susceptible to pesticide phytotoxicity, although some species are more tolerant than others. In an Oregon nursery, the herbicide DCPA produced stem swelling and stunting of Douglas-fir and true fir seedlings, but did not affect five different species of pines. Western larch has been shown to be highly susceptible to phytotoxicity from many of the pesticides used in forest nurseries.

There is evidently genetic variation in susceptibility to pesticide phytotoxicity. Pesticide damage was definitely related to seed source when a fumigant inadvertently drifted into a white pine progeny trial; the severity of injury was clearly related to the genetic origin of the seedlings (Figure 30-1). Phytotoxicity symptoms have been reported to vary considerably



B

between different seedlots of Douglas-fir and true fir seedlings.

The occurrence and relative severity of phytotoxicity damage is also related to the developmental stage of the seedlings. Young germinants or actively elongating shoots are much more sensitive than hardened or dormant stock. Some selective herbicides, such as oxyfluorfen, have caused needle twisting when applied to actively growing shoots, whereas applications only a few weeks later in the growing season were safe.



C

**Figure 30-2. Foliar symptoms due to pesticide phytotoxicity: (A) foliar discoloration and distortion, (B) needle tip burn, (C) needle twisting.**

### **Symptoms**

Pesticide phytotoxicity and other types of chemical injury, including damage from air pollution and fertilizers, can often be distinguished from biological pest problems by the pattern and timing of symptom development. Pesticide damage symptoms occur all at once and often have a regular distribution in the seedbed, whereas symptoms caused by pathogens usually develop over an extended period of time and occur in random or grouped patterns.

Pesticide phytotoxicity can be expressed by a number of different foliar symptoms, including discoloration, needle tip burn, and needle twisting (Figure 30-2), as well as swelling of the stem (Figure 30-3), and stunting and mortality of

seedlings (Figure 30-4).

Phytotoxicity symptoms are similar for many different pesticides, although some chemicals produce unique symptoms that have particular diagnostic value. Herbicides with hormone-type actions produce easily identifiable symptoms, such as the leaf cupping and needle twisting that results from drift of the extremely volatile 2,4-D. Simazine and atrazine produce chlorosis between the veins and on the margins of hardwood leaves, and needle tipburn in conifers. Amitrole destroys chlorophyll, so that affected foliage is whitish or even pinkish in color.



Several herbicides have been reported to arrest seedling root development, often resulting in abrupt cessation of root growth at a uniform depth in the soil (Figure 30-5). The herbicide napropamide snubbed off the roots of western larch about 1 inch below the ground line, and most symptomatic seedlings eventually died.

## Predisposing factors

Weather events often predispose seedlings to pesticide phytotoxicity. With nonselective pesticides, temperature inversion may trap fumigant gases near the ground, causing them to accumulate in low areas. Soil fumigants should be applied only when temperatures are warm enough that the vapor can completely disperse within the recommended period after application. Heavy rainfall can cause surface runoff of water-soluble nonselective

**Pesticide phytotoxicity  
appears:  
All ages  
Any time after application**

pesticides into crop areas.

Even normally safe selective herbicides can cause injury under predisposing weather conditions. Sprays and soil drenches should not be applied when the soil is extremely dry and the seedlings are under moisture stress. The fungicide captan has caused stem damage and even mortality to young, succulent seedlings when applied under hot, dry conditions. On the other hand, cool weather after application is reported to increase the incidence of DCPA phytotoxicity.

## Loss potential

Losses caused by pesticide

phytotoxicity can range from imperceptible reductions in growth to mortality of seedlings. The exact amount of loss varies considerably; it depends on weather conditions, the susceptibility of the crop, and the size of the affected area. As an example, DCPA phytotoxicity affected 50 to 80 percent of the Douglas-fir and true fir seedlings in an Oregon nursery. At an Idaho nursery, mortality from napropamide phytotoxicity caused a 50-percent reduction in seedbed density of western larch, and the associated stunting reduced the number of shippable seedlings by almost 90 percent. Minor growth losses are difficult to assess, but damaged foliage can cause seedlings to be culled on the grading table.

## Management

Prevention is the only management option for controlling phytotoxicity losses. Nursery managers should consider the following measures:

- 1. Train pesticide applicators.** Pesticide applicators are required by state law to attend regular training sessions. Nursery managers should ensure that all employees who handle pesticides become certified. In-house pesticide training should also be regularly scheduled so that applicators can be informed of local weather conditions and trained in proper calibration and use of the nursery's specific application equipment.
- 2. Keep accurate pesticide application records.** Pesticide applicators should be required to fill out predesigned application forms that ensure that the same relevant information is always recorded. This information should include date, pesticide used, area treated, prescribed application rate, actual application rate, and weather and soil conditions. In case suspicious symptoms show up, these applica-



**Figure 30-3. Abnormal swelling of the stem due to pesticide phytotoxicity.**



**Figure 30-4. Mortality and stunting caused by a residual soil herbicide.**

tion records can be checked to see if phytotoxicity could be the cause. The exercise of calculating the actual pesticide application rate each time also serves as a cross-check of whether the application equipment is operating properly.

- 3. Monitor weather conditions before and during pesti-**

**cide applications.** Nursery managers should check nursery weather stations and consult local forecasts before applying pesticides. Critical operations, such as seedbed fumigation, should never be attempted if marginal weather conditions exist or are forecast. The early morning is usually the best time to apply liquid sprays because winds are minimal and temperatures near the soil surface are low.

**4. Leave control plots.** Phytotoxicity can be very difficult to diagnose because similar symptoms can be caused by other factors. Growth losses due to phytotoxicity are almost impossible to identify. Nurseries, therefore, should establish a policy requiring that untreated control plots be established each time a pesticide is applied. These plots should be clearly marked and regularly checked during the growing season so that slow-to-develop symptoms can be identified. Control plots can also be used to illustrate the effectiveness of a specific pesticide treatment, documenting differences in seedling growth or yield. These data are essential for calculating cost-benefit ratios when determining the economics of pesticide use.

**5. Conduct nursery-specific phytotoxicity trials.** Because soil type, local climate, and seedling genotypes differ among nurseries, each nursery should test new pesticides before beginning large-scale operational use. Many phytotoxicity incidents could have been prevented if the nursery had first tested the pesticide under its own unique conditions.

**6. Isolate pesticide-sensitive species.** Species that are especially susceptible to chemical injury—western larch, for example—should be planted in sections of the nursery that are isolated from production areas where pesticides will routinely be used.

**7. Test the soil for persistent pesticides in newly developed seedbeds.** If pesticide contamination or residues are suspected, a sample of the soil can be enclosed in a glass jar and sown with seeds of a fast-growing plant, such as radish or lettuce. If the seeds fail to germinate or if the germinants exhibit abnormal growth, additional tests or corrective treatments are warranted. If the soil was recently fumigated, allow more time for the fumigant to escape. In the case of persistent nonselective pesticides, treating the soil with activated charcoal may be effective.

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**Figure 30-5.** Bean and Douglas-fir showing abnormal root development. The healthy bean plant on the far left shows normal root elongation. Beans, highly sensitive to the herbicide, were sown as an assay to detect residual chemical.

## CHAPTER THIRTY-ONE

# Salt Injury

Thomas D. Landis  
David E. Steinfeld

A salt is a chemical compound that breaks down in water into electrically charged particles called ions. In a soil system, soluble salts are defined as those inorganic chemicals that are more soluble than gypsum ( $\text{CaSO}_4$ ). The principal soluble salts in the soil contain the cations sodium ( $\text{Na}^+$ ), calcium ( $\text{Ca}^{2+}$ ), and magnesium ( $\text{Mg}^{2+}$ ) and the anions chloride ( $\text{Cl}^-$ ), sulfate ( $\text{SO}_4^{2-}$ ), and bicarbonate ( $\text{HCO}_3^-$ ). Other ions, such as carbonate ( $\text{CO}_3^{2-}$ ) and boron ( $\text{B}^{3+}$ ), can sometimes be present.

Salts may be either damaging or beneficial depending on the characteristics of the specific chemicals involved and the total salt concentration. Ordinary table salt ( $\text{NaCl}$ ) is toxic to plants, whereas magnesium sulfate ( $\text{MgSO}_4$ ) can be used as a fertilizer. Soluble salts in forest nurseries can come from several sources. They can occur naturally in the soil, or they can be introduced in saline irrigation water, through overfertilization, or from salt-contaminated mulches.

Soluble salts can injure woody plant seedlings in four different ways, depending on the total salt concentration and the specific salt ions involved:

- A high total soluble-salt concentration can cause an osmotic effect—the salt ions can chemically reduce the water available to the seedling.

- The permeability and water infiltration rate of the soil are reduced because high relative concentrations of sodium salts cause soil particles to disperse.

**Table 31-1. Guidelines for interpreting result of electrical conductivity (EC) tests of irrigation water and soil. Ratings, expressed in microSiemens per centimeter (mcS/cm), show adequate, marginally high, and excessive levels of salinity.**

|                              | ADEQUATE     | MARGINALLY HIGH | EXCESSIVE |
|------------------------------|--------------|-----------------|-----------|
| Irrigation water             | 0 to 500     | 500 to 1,500    | >1,500    |
| Soil<br>(saturation extract) | 750 to 2,500 | 2,500 to 4,000  | >4,000    |

- High levels of certain ions, including sodium, chloride, and boron, are directly toxic to plants.

- An imbalance of salts that are also nutrients, such as calcium, can reduce the availability of other mineral nutrients, such as iron or phosphorus.

Obviously, the diagnosis of salt injury is a complex subject. Only the osmotic effect of high total salt levels will be considered here, because that is the most widespread problem. The other three specific ion effects cause problems on a more local basis. Because the diagnosis and treatment of these problems is more complicated, additional information can be obtained from the references supplied at the end of this chapter.

High soluble salt concentrations reduce the osmotic potential, and therefore the total water potential, of the soil solution. This, in turn, reduces the water that is available to the roots of seedlings, causing a type of physiological drought. This

osmotic effect is particularly damaging during seed germination and seedling emergence, but can also cause problems with larger stock if the soil is allowed to dry out excessively.

## Occurrence: species and season

All seedlings are susceptible to salt injury to some degree. Germinants and young seedlings are particularly vulnerable because of their succulent nature, whereas older seedlings and other stock types, such as transplants, are more tolerant. Small-seeded or slow-growing species like spruces and firs are often more susceptible because they take longer to grow out of the young, succulent stage.

Whereas the salt tolerance of most agronomic crops has been established, that of many species of Pacific Northwest forest tree seedlings has never been determined. Most commercially produced nursery stock should be considered susceptible, however. Pine seed-



A



B



C

lings are generally more salt-tolerant than very susceptible species such as Douglas-fir and spruces. There is variation within *Pinus*, however; lodgepole pine is less tolerant than ponderosa pine. Susceptibility to salt damage does not change during the growing season, although there is more potential for injury when seedling tissue is succulent or during periods of high moisture stress.

## Symptoms

In germinating seeds and emerging seedlings, mortality caused by salt injury may be mistakenly attributed to damping-off. In larger seedlings, visual symptoms of salt injury will vary among species, stock types, and ages of the seedlings. Salt-affected



D

**Figure 31-1. Foliar symptoms caused by salt injury: (A) chlorosis, (B) and (C) scorched needle tips, and (D) mosaic growth pattern of normal and stunted seedlings.**

**Salt injury may be confused with**  
**Damping-off**  
**Pesticide damage**  
**Mineral nutrient problems**

seedlings may exhibit foliar chlorosis, scorched needle tips or leaf margins, or a patchy, mosaic

growth pattern of normal and stunted seedlings (Figure 31-1). Seedlings growing in saline soils often do not develop fibrous root systems. Soils dominated by calcium salts often exhibit white surface crusts (Figure 31-2) that effervesce when tested with a drop of dilute acid (Figure 31-3). Soils with a high proportion of sodium salts, on the other hand, are blackish in color and have a slippery texture.

Salt injury can not be identified by visual symptoms alone. Accurate diagnosis requires a comprehensive evaluation, including chemical analysis of soil, irrigation water, and seedling tissue. The best measure of the total soluble-salt level in irrigation water or soil is the electrical conductivity (EC) test, which can be performed with a conductivity meter. Because salt ions conduct electricity, an EC test can measure the salt concentration

in an aqueous solution. The EC of irrigation water can be tested directly. The EC of a soil sample is measured using a saturation extract, which is obtained by adding enough distilled water to soil to form a thick paste and extracting the soil solution. EC tests can measure only total soluble salts, not levels of individual salt ions. Table 31-1 gives general guidelines for interpreting EC test results. SI units are microSiemens per centimeter (mcS/cm), which are equivalent to the older units of micromhos per centimeter (mcmhos/cm). Soil samples should be collected at a number of different depths but especially from the surface, because salts often wick to the surface when water evaporates from the soil.

**Salt injury symptoms  
appear:  
All ages  
Any time throughout rotation**

### Predisposing factors

Salt injury can occur without any predisposing stress factors, although hot, dry weather can increase the potential for damage. Poor irrigation practices that allow the soil to dry out will concentrate the salts in the soil solution. In particular, brief irrigations used for cooling seedlings during hot weather can increase the salinity in surface soils.

Poor soil-management practices can lead to accumulation of salts in the seedling root zone, especially in fine-textured soils. Improper or excessive soil cultivation can break down soil structure, reducing porosity and inhibiting water infiltration and drainage. Repeated use of heavy equipment in seedbeds can produce impermeable soil "pans" within the soil profile, further restricting drainage.



**Figure 31-2.** Soils dominated by calcium salts often exhibit white surface crusts.



**Figure 31-3.** The presence of excessive calcium salts can be confirmed by placing a drop of dilute acid on the soil to see if effervescence occurs.

### Loss potential

Mortality during the germination period can be significant, but these losses often go unnoticed, especially if the damage is done before the seedlings emerge. With older seedlings, growth losses are difficult to quantify because the growth rate may be significantly reduced before the stunting be-

comes evident. Stunted seedlings result in increased cull rates, particularly when they are scattered throughout the seedbeds, because they make it necessary to lift and grade entire seedbeds.

## Management

### CULTURAL

The best solution is to avoid salinity problems in the first place by judicious selection of nursery sites. Soils that are inherently saline can be identified during site selection, but even initially productive nursery soils can be ruined with moderately saline irrigation water. Therefore, both soil and irrigation water should be evaluated for salinity problems; not only for total soluble salts but also for the relative concentration of specific salt ions. Soil permeability and porosity should also be tested when evaluating nursery sites, because they determine the leaching potential of the soil.

In established nurseries, managers should conduct intensive soil surveys of their seedling production areas to identify the least-saline soils for producing salt-sensitive species. More-tolerant species and stock types, such as transplants, can be planted in the more-saline nursery blocks.

The only way to remove soluble salts from the soil is to leach them from the soil profile with deep irrigation—applying large amounts of water to the soil to dissolve the salts and carry them down below the root zone. Obviously, leaching will be more effective if irrigation water with a low salt content is used. These irrigation treatments should be scheduled during the fallow year, after the soil has been deep-ripped to break up any impermeable hardpan layers. Incorporation of relatively large amounts of organic matter will help increase the porosity of most soils, and gypsum amendments can improve soil structure in high-sodium soils. Deep irrigation is particularly effective in reducing the salt concentration in surface soil layers during the critical seed germination and emergence period. This treatment must be monitored carefully, however, to ensure that soils do not become waterlogged. Once the seedlings are established, deep

irrigation is more difficult to accomplish, but it can be scheduled immediately after wrenching when the soil is most porous.

Mulches may be used to decrease evaporation from the soil surface and keep salt crusts from developing. Light-colored mulches are better because darker mulches absorb heat, which can girdle succulent seedlings. Hydromulch is effective when applied immediately after sowing. A thin layer of sawdust is also an effective mulch; it can be applied either to the sown seedbed or over existing seedlings. Sand mulches should be tested to make sure they are not calcareous.

Because inorganic fertilizers are also salts, only products with a low salt index should be considered for use on saline soils. A series of small fertilizer applications is less likely to cause injury than one or two large applications. Organic fertilizers such as sludge or animal manures may contain high salt levels and should be carefully analyzed before they are purchased for use in forest nurseries. Even sawdust can be saline if the logs were stored in salt water for long periods.

### CHEMICAL

There is no simple way to treat the soil chemically to correct a salinity problem. Sodium-affected soils can be treated with gypsum amendments of about 22.4 mt/ha (10 tons/ac) to improve soil porosity and prepare it for leaching.

If the soil contains insoluble calcium carbonate, that salt can be chemically converted to the more soluble gypsum with amendments of elemental sulfur at rates of 560-1,120 kg/ha (500-1,000 lb/ac). These sulfur treatments will also lower the soil pH. Because it is slow to react, sulfur should be applied at least a year before the seedling crop is sown.

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## CHAPTER THIRTY-TWO

# Soil Compaction

David E. Steinfeld  
Thomas D. Landis

Although most people think of soil as a solid material, it actually consists of a mixture of solid particles and the pores that exist between them. Biologically speaking, the porosity of a soil is just as important as its solid particles because the pores conduct the water necessary for plant growth and the air necessary for root and microbial respiration.

**Soil compaction symptoms appear:**  
**All ages**  
**Any time throughout rotation**

Soil compaction can be defined as the decrease in pore space—most importantly, a decrease in the size of the pores—through either natural or cultural means. Although the soil in the entire plow layer will become compacted to some degree, the adverse effects of compaction can often be traced to specific compacted layers, or "pans," which usually develop at certain depths in the soil. Even though soil pans often form below the normal rooting zone, they restrict soil drainage, which in turn adversely affects the soil layers above.

The physical effects of compaction include an increase in bulk density and soil strengths and a decrease in drainage (permeability), water-holding capacity, and air movement within the soil. These physical effects cause a degrada-



**Figure 32-1.** Soil compaction can result in poor root development. In this seedling, fine roots have failed to develop.

tion in the biological and chemical properties of the affected soils. Restricted exchange of air and water reduces seedling root growth and encourages pathogenic organisms at the expense of favorable microorganisms such as mycorrhizae. After extended periods of compaction, the conditions in poorly aerated soils can reduce mineral nutrient uptake. Alone or in combination, these compaction-induced changes in soil properties can impair the growth of forest tree seedlings.



**Figure 32-2.** Roots killed by soil compaction. A soil pan that obstructs drainage causes an anaerobic environment, making root survival impossible. Dead roots are dark gray or black.

## Occurrence: species and season

All seedling species and stock types are susceptible, and because soil compaction affects root function and growth, seedlings of all ages are affected. Some species are more tolerant of poorly drained soils than others. Three-needle pines and cedars are most tolerant, seven-needle pines are less tolerant, and Douglas-fir and true firs are most susceptible. The occurrence of compaction injury and the degree of damage depend more on soil characteristics and nursery



**Figure 32-3. Diseased roots also indicate soil-compaction problems. *Phytophthora* root rot is often found in areas where drainage is poor because soil is compacted.**

cultural practices than on species of seedling.

## Symptoms

Compaction can be diagnosed by observing seedling symptoms and by testing the soil. Because compaction primarily affects the root system, many of the initial symptoms are hidden underground. Problems with reduced seedling vigor and growth rate soon develop, however, and many of the visible shoot symptoms can be attributed to earlier root problems. Specific symptoms of compaction are poor root development, particularly of the fine roots; dead and diseased roots; and chlorotic and stunted seedlings.

### Poor root development.

Compaction increases soil strengths, making it more difficult for seedling roots to penetrate compacted soil. Seedlings grown in compacted soils generally have poorly developed or distorted root systems. They often lack fine roots

(Figure 32-1) and their root volumes are generally smaller than those of seedlings from surrounding uncompacted areas.

### Dead and diseased roots.

Roots that die as a result of compaction are dark gray or black in color and are often found at a uniform depth in the soil. This zonal pattern is caused by a soil pan that prevents drainage, resulting in a continuously saturated, anaerobic environment in which survival of roots is impossible. Diseased roots often indicate a soil compaction problem (Figure 32-2). Such root pathogens as *Pythium* and *Phytophthora* flourish under the saturated soil conditions created by compaction (Figure 32-3).

### Chlorotic and stunted seedlings.

Seedlings grown in compacted soils often become chlorotic and stunted in their first growing season (Figure 32-4). In extreme cases the new needles on the terminal may appear burned. Compaction symptoms generally occur in regular patterns across the seedbeds. These patterns can sometimes be correlated to the

ways in which nursery equipment has been used—the direction of plowing or ripping, for example.

There are many ways to assess compaction in nursery beds, some of which require special equipment and training. The following techniques are the most practical for the nursery manager:

*Shovel penetration*—In this quick test, a shovel is used to determine whether compaction is present. If the weight of a person standing on a shovel in a moistened seedbed is not enough to make the shovel pass easily through the soil profile, compaction is likely. The main factors affecting shovel penetration are the moisture content of a given soil and the amount of pressure applied to the shovel. Any measurement of the soil strength should be done when the soil is uniformly moist (the drier the soil, the harder it is to penetrate) and when its moisture content is known, so that comparative measurements can be taken. Establish a standard sampling time by taking measurements 1 to 2 days after a deep irrigation. This should assure a similar soil moisture content for each test.



**Figure 32-4. Poor 1+0 seedling growth due to soil compaction. Tall seedlings are growing in uncompacted soil. Stunted seedlings were sown in soil that was inadequately cultivated.**



*Cone penetrometer*—The shovel test is quick but not highly accurate, since it is difficult to quantify the pressure on a shovel. A cone penetrometer measures the force needed to push a standard rod with a sharp tip through the soil. The penetrometer is a very reliable tool for quickly assessing whether there is a compacted layer in the soil. Moreover, because several readings can be made only inches apart, the penetrometer can be used in seedbeds with little disturbance to seedlings. Because penetrometer readings are relative, nursery managers must establish standards for their own soil conditions. This can be done by taking readings in areas where seedlings are showing visual symptoms of compaction and comparing them with readings taken in soil of the same type in which healthy seedlings are growing. Using these standards, managers can identify compacted areas before seedlings begin to show symptoms. If symptoms are already evident, a penetrometer can determine whether compaction is the problem.

**Soil compaction may be confused with:**  
**Phytophthora root rot**  
**Nematode damage**  
**Mineral nutrient problems**

*Ring infiltrometer*—Poor drainage due to compaction can be diagnosed with a ring infiltrometer, which is a circular metal ring or tube that is pushed into the soil and filled with water. The time it takes for the water level to drop a designated distance inside the ring is an indication of the permeability of the soil. The slower the water moves into the soil, the more serious the compaction. A related but simpler technique is to dig a series of small holes at several depths in the test area, fill them with water, and monitor the water level.



**Figure 32-5.** Soil type, moisture content, and equipment use are all factors in soil compaction. Lifting seedlings when soil is too wet can be particularly damaging to soil.

## Predisposing factors

Compaction in agricultural soils is primarily a function of soil type, soil moisture content, and equipment use. Each soil has its own characteristic response to compactive forces which depends on soil mineralogy and texture and the type and amount of organic matter in the soil. Soil mineralogy and texture are very important factors. Finer-textured silts and clays are more subject to compaction than coarser-textured sands. Because soil structure is improved with humus—the end product of the decomposition of organic matter—soils containing high organic matter levels are resilient and resist compaction.

Soil moisture content is one of the most critical factors. Within a given soil textural class, compaction increases with moisture content. The timing of heavy equipment use is also critical; many compaction problems result from using tractors and other equipment during excessively wet periods, especially during the winter lifting season (Figure 32-5).

## Loss potential

Compaction is rarely a direct cause of mortality, but its effects can severely reduce the number of shippable seedlings per unit area of seedbed. Growth losses due to compaction result in a higher cull rate and poorer overall stock quality. Compaction losses generally show up on the grading table in the following cull categories: stunted and chlorotic seedlings, sparse or diseased roots, and mechanical damage to root systems.

Stunting and chlorosis caused by soil compaction begins in the first year and often carries over into the second. Growth losses can be considerable in 1+0 stock types, but the impact on 2+0 seedlings can be minimized with corrective cultural practices.

Poorly developed root systems are a result either of the adverse growing environment or the activity of root-disease organisms in compacted soils. Because of their minimal root systems and related poor root-to-shoot ratios, seedlings from compacted soils are normally culled during the grading process.

Moreover, seedlings growing in compacted soils often suffer extensive root damage when they are harvested. Depending on the amount of compaction present and the type of lifting method used, such damage can result in very high culling losses.

Root injuries, especially minor root infections, are often difficult to recognize on the grading table. If these compaction-related problems are not identified during grading and packing, seedlings with inferior root systems will perform poorly in the field when outplanted.

## Management

The best management practice is to prevent compaction in the first place. Nursery managers can use the following techniques to prevent compaction or minimize its effects:

### 1. Selection of seedbeds.

Growers should identify and map compacted areas, leaving them unplanted until corrective action can be taken. Stock types that are most sensitive to compaction, such as 1+0 seedlings, should be planted in the best soils available.

### 2. Maintenance of organic matter.

Green-manure crops, cover crops, and organic-matter amendments can increase, or at least maintain, the level of organic matter in the soil.

### 3. Subsoiling.

This specialized form of tillage, also called deep ripping or chiseling, breaks up compacted layers at depths greater than 12 inches. The tool most commonly used for this operation is the subsoiler, which is designed to fracture compacted soil layers or pans. Soil moisture content is critical for successful subsoiling. If conditions are too wet, the subsoiler tines will slice through the soil instead of shattering it. If it is too dry, blocks of compacted soil will resist being fractured. The proper soil moisture level will vary

with soil type; each nursery must determine the proper conditions for its own soils. A minimum of two passes should be performed, with the second pass at an angle of 30 to 45 degrees from the first. The tines should not be drawn too deeply on the first pass. It is more effective to set the tines relatively shallow initially and then lower them on the second and third passes.

### 4. Minimizing the use of heavy equipment.

In new or recently subsoiled seedbeds, use of heavy equipment should be kept to a minimum. In fact, the first three passes of a heavy machine over an uncompacted nursery bed are the most damaging. The type of equipment used is also important. Equipment operators should be instructed always to use the smallest tractor that will perform the task. Dual-tire and crawler tractors should be used whenever possible because they compact the soil less than single-tire tractors. The best time to operate heavy equipment on most soils is when they are dry. Unfortunately, certain nursery operations, such as harvesting of seedlings, must be conducted during the winter season when soils are wet and most subject to compaction.

**5. Wrenching.** After the seedlings emerge, compaction can be mitigated to some extent by wrenching the seedbed. Wrenching consists of drawing a horizontally mounted angled blade under the seedlings, which shatters the soil around their roots. As with subsoiling, the proper soil moisture content and blade depth must be tested at each nursery to achieve the proper results. Wrenching is the only practical means of reducing compaction after the crop has been sown.

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## CHAPTER THIRTY-THREE

# Principles of Integrated Pest Management

Everett M. Hansen, Philip B. Hamm, Sally J. Campbell

Most forest tree nurseries operating in the Northwest today consistently produce a profitable crop of high-quality trees. Successful nurseries integrate pest-management activities into the overall nursery operation. Four features of effective pest management are: 1) clear nursery goals, 2) a planned decision-making process, 3) realistic damage thresholds, and 4) a choice of responses.

## Goals

Nursery goals should be clearly defined and comprehensive. Seedling quality, stock type, and quantity will be at the top of most lists of goals. Cost efficiency will also be a high priority. Issues of environmental and human safety must be addressed, as well as concerns about long-term productivity of the nursery soil. The specifics will vary between nurseries and sometimes between crops. Nevertheless, nursery goals provide the basis for good decision-making.

Nursery production is a carefully orchestrated process. Many pest-management measures are taken as part of the overall schedule of activities. Pest outbreaks are not always predictable, however, and advance planning can make the difference between a successful control operation that is consistent with overall nursery goals, and a hasty reaction that tries to solve an immediate problem only to create new ones later on.

## The decision-making process

An important element in consistent pest control is the decision-making process: how the manager decides if a pest needs to be controlled, when to take action, and what methods to use. The process should be developed and made a part of the formal management policy so that it can survive personnel changes, and careful records should be kept so that methods of control can be evaluated or more-appropriate responses chosen in the future.

Pest-management decisions should be based on documented pest status, including historical occurrence, field monitoring data, and climatic or other predictive factors; the analysis of treatment options; and the potential of these elements to affect the nursery's goals. Such a process is illustrated in a flow chart (Figure 33-1) to help nursery managers arrive at sound decisions.

## Damage thresholds

Pest management is enhanced when potential pests and options for managing them are identified before major losses occur. Nursery pests are always present, but as long as their numbers are low, damage is usually insignificant. Since all pest-management operations carry costs and risks, it is important to decide on realistic thresholds for pest populations and damage in order to guide effective treatment.

## A choice of responses

Many pest problems can be prevented by careful planning or stopped when detected early. Pre-planned options afford a reasoned selection of appropriate pest-management tools that are consistent with the nursery's goals. Most pest problems can be addressed by a range of cultural, chemical, and biological control tactics.

These four features of successful nursery pest management—clear goals, planned decision-making, realistic damage tolerances, and a choice of appropriate responses—along with systematic monitoring and evaluation, are the components of Integrated Pest Management (IPM). IPM is nothing more than a common-sense, systematic approach to problem-solving in the nursery. The following sections describe procedures to monitor various pests and discuss the range of available pest-management tools.

## Monitoring of pests

Pest and damage monitoring provides both early warning of pest problems and a measure of the efficacy of control actions. Data accumulated systematically over several years can be the basis for predicting the impact of damage from pests and for setting thresholds of acceptable damage. Monitoring can consist of tracking the damage caused by the pest, the actual pest population, or a combination of both.

### NURSERY GOALS

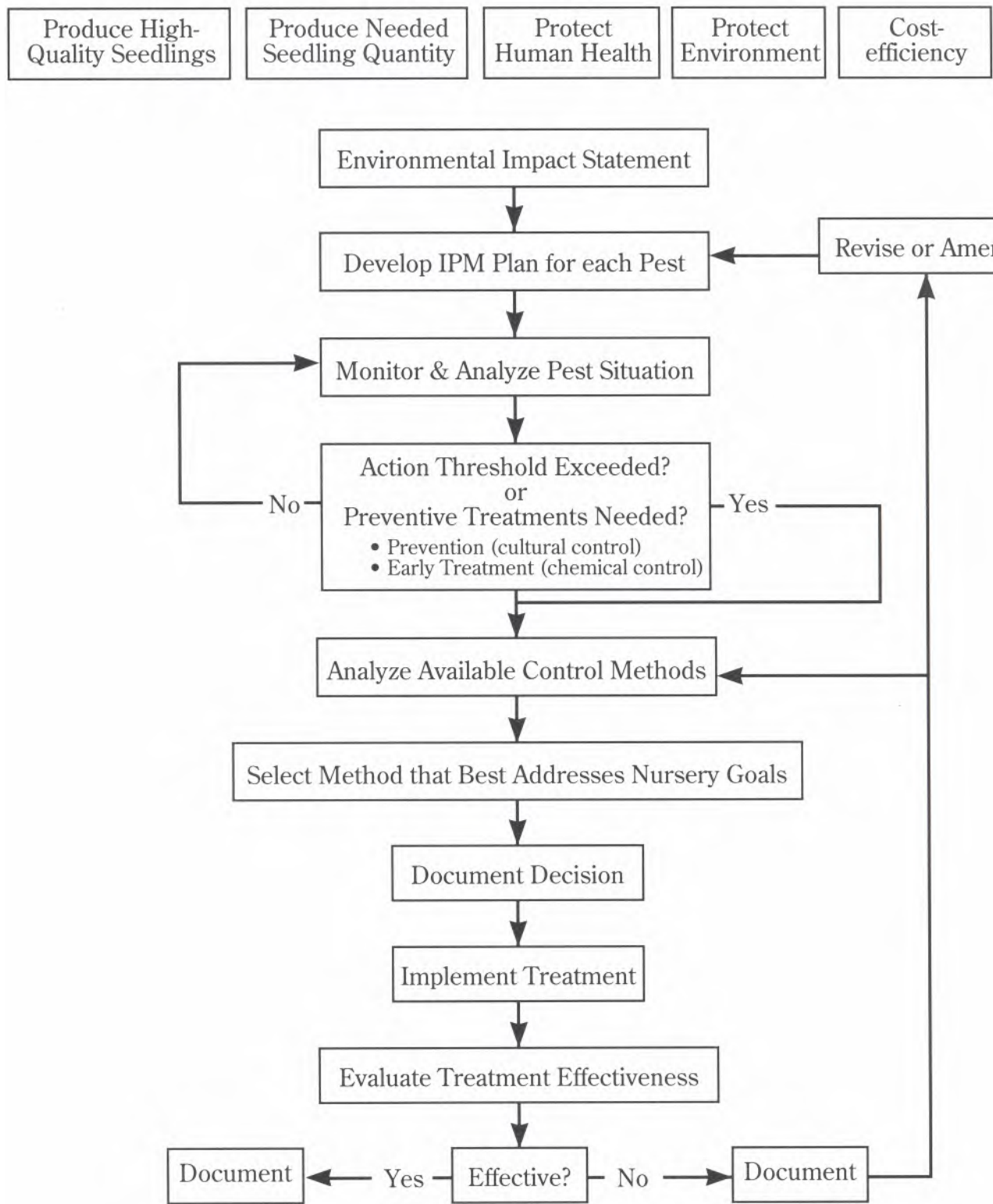


Figure 33-1. A flow chart to guide pest management decisions toward nursery goals. Taken from the Final Environmental Impact Statement, U.S. Department of Agriculture, Forest Service, Nursery Pest Management, Pacific Northwest Region, October 1989.

A good damage-monitoring program requires people trained to recognize early symptoms of trouble. It should also include systematic field-survey, record-keeping, and evaluation procedures. The program should cover all parts of the nursery regularly, with more-frequent attention given to areas with chronic problems and beds where particularly susceptible tree species are grown.

Knowing when and where to look and what to look for is critical to a successful program. Expert diagnostic help is available (see Introduction) but consistent success will depend on skilled nursery personnel. Private consultants can be hired to take over all or part of the process.

#### INSECTS

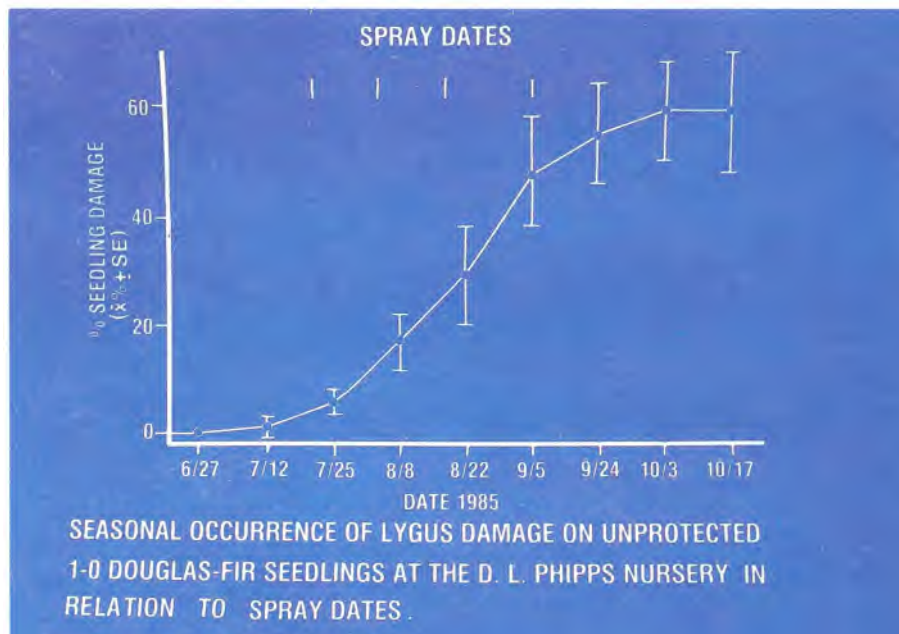
In the Pacific Northwest, most detection surveys for insect problems should be conducted between May and September, when insects are most abundant and active. This handbook gives general guidelines on when to look for insect activity. However, the onset of problems from a given insect typically varies by several weeks over large geographic areas.

Foliage-feeding insects such as cutworms and lygus bugs are best detected by their damage. Sweep nets can also be used to gather insects from seedling foliage. The onset of lygus bug damage symptoms in 1+0 Douglas-fir can be used to time insecticide applications.

It is generally not feasible to monitor directly for root- and stem-feeding larvae such as white grubs, root weevils, or cranberry girdlers because numbers are low and populations are scattered. Often the first indication of root damage is the death of seedlings during the fall and winter months. Although cranberry girdler larvae are difficult to monitor, the pest can be trapped in the moth stage and moth populations assessed as an indicator of future damage by larvae (Figure 33-2).

#### NEMATODES AND FUNGI

Diseases caused by soilborne



**Figure 33-2.** Cranberry girdler trap catch data for 2 years in a Pacific Northwest nursery. This information reflects moth abundance and defines the period for effective insecticide applications. Weyerhaeuser Co. photo.

fungi and nematodes can be monitored by looking for typical damage on the seedling or by assaying the soil for the pest. Diseases such as gray mold, caused by fungi that are not soilborne, are usually monitored by observing the damage or signs of the pest on aboveground parts of the seedling.

Soil assay services may be employed to determine populations of nematodes and of the pathogenic soilborne fungi *Pythium*, *Fusarium*, and *Macrophomina*. Reliable assay results depend on an adequate sampling strategy and careful handling of samples.

A soil sampling tube is the quickest, easiest, and most reliable tool for collecting samples (Figure 33-3). The soil should be moist but not saturated. Soil cores should be taken to a depth of 12 inches. The sample that is submitted for analysis should be made up of a number of subsamples and should represent a single soil type and crop history collected from no more than one-eighth of an acre. Generally, the greater the number of subsamples, the more accurate the results. Subsamples should be thoroughly

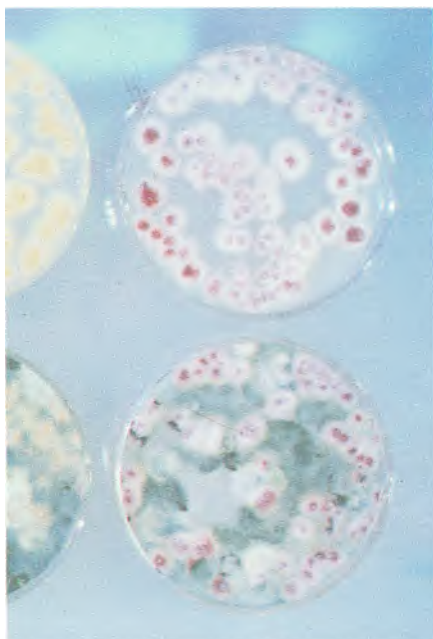
mixed in a clean container, then 1/2 liter (1 pint) of the mixture transferred into a plastic bag. Always pack soil bags in boxes before shipping them to the laboratory.

Improper handling of samples kills nematodes and fungi and will yield an inaccurate analysis. Do not leave samples in direct sunlight, in the trunk of a car, in the back of pickup, or in a freezer. While samples may be stored at 10-14 degrees C (50-58 degrees F) for a short time, or even longer at cooler temperatures, they should be sent in for analysis as quickly as possible.

*Nematodes*—An assay for nematodes will help both in diagnosis of current problems and prediction of future problems. Comparative samples must be collected from healthy and symptomatic areas. Both the soil and the roots of live symptomatic seedlings should be sampled. Dead seedlings should not be included in the sample because nematode populations will have declined due to the lack of food. If nematode numbers prove to be several times higher in the sympto-



**Figure 33-3.** Collecting soli for laboratory assay.



**Figure 33-4.** Counting *Fusarium* colonies, like these, determines the number of fungal propagules present in a soil sample.

matic area than in the healthy area, nematodes are the likely cause of the poor growth. If the populations are high in both areas, nematodes may still be involved as secondary agents and nematode control should be considered. Because nematode damage generally occurs in proportion to population, nematode assays are also useful for predicting future losses. However, since numbers are usually low in nurseries that fumigate regularly, predictive sampling may not be routinely needed.

*Soil fungi*—*Samples* collected for nematode assays may also be used to assay for soil fungi. Populations of *Pythium*, *Fusarium*, and *Macrophomina* can be determined by dilution-plate assays (Figure 33-4). As with nematode assays, information about soil fungi can provide a useful measure of current conditions and a gross indicator of future problems. Soil-fungi assays can determine the effectiveness of fumigation and may help diagnose damping-off problems in the early stages, thus allowing timely and specific response with fungicides. However, because fungal populations vary widely over short distances, because actual damage to seedlings requires both the presence of the fungus and specific environmental conditions which are poorly understood, and because pathogenicity of propagules is not known, it has not been possible to predict future disease losses reliably from soil-assay data. As more nurseries keep careful records, we will be better able to use soil assays to help manage certain pest problems.

## Pest management

This section discusses the general methods for pest management: cultural, chemical, and biological control. Cultural control refers to the routine nursery operations that help reduce losses to pests. Chemical control involves the application of pesticides to the soil, seeds, or seedlings to reduce pest damage. Bio-

logical control involves the use of living organisms, generally predaceous insects or nonpathogenic fungi or bacteria, to manage pest populations. Few artificially introduced biocontrol agents are available for control of nursery pests in bare-root nurseries in the Northwest.

## CULTURAL CONTROL

Cultural control of nursery pests provides the first and often the most effective defense. Both fungal and insect pests of nursery crops have critical environmental and nutritional requirements for feeding, reproduction, and infection of the host. Nursery cultural practices can modify these conditions to the disadvantage of the pest. Cultural control can limit pest populations or damage directly, by removing or suppressing the pest, or indirectly, by altering its environment or food supply. Five categories of cultural control will be considered: 1) soil management, 2) water management, 3) practices affecting seedling growth, 4) sanitation, and 5) growing resistant seedlings.

*Soil management*—*Physical*, chemical, and biological properties of nursery soil are critical to seedling health. Well-drained soils allow free root growth and limit opportunities for the water molds (*Pythium* and *Phytophthora*) to increase to damaging levels. Any place that collects standing water during heavy rains or irrigation is a potential trouble spot. It is often necessary to crown the fields and install underground drainage lines. Raised beds are usually beneficial as well. Soil compaction causes or aggravates many water problems. Minimizing equipment use, especially on wet soils, reduces compaction, and deep ripping can restore drainage in compacted soils. It may be necessary to stop using chronic wet spots for nursery production.

Organic matter in the soil is important for soil tilth and as a food source for microorganisms, both beneficial and damaging. Recent experiments show that most cover

crops encourage soil pathogen populations, especially in unfumigated fields (Figure 33-5). In these experiments, no cover crop at all (bare fallow) gave the best subsequent conifer crop. Sudan grass harbored higher pathogen levels than bare fallow and resulted in increased seedling mortality. Legume cover crops (beans or peas) were most detrimental to the health of the subsequent seedling crop (Figure 33-6).

Other work, however, suggests that certain *Brassica* cover crops may reduce *Fusarium* and *Pythium* populations. Sawdust amendments have been effective in controlling seedling diseases in both eastside and westside nurseries. As yet there are no universal recommendations on the use of organic matter. However, it is clear that nursery managers should critically evaluate the addition of any cover crop or soil amendment.

**Water management**—Water is widely used as a cultural tool to limit problems caused by high summer temperatures and by freezing temperatures in late fall or early spring. Too much water, however, saturates soils, which denies oxygen to roots and allows soilborne fungi to reproduce and spread. Water on foliage for extended periods of time allows stem and needle pathogens such as gray mold to germinate and cause infection, and excessive irrigation washes off protective fungicides.

The art of water management starts with good soil drainage (Figure 33-7). Irrigation needs should be coordinated with summer cooling requirements and pesticide application schedules. Both the total amount of water applied, and the frequency and duration of the times the foliage is wet, should be kept to a minimum. Paying attention to details like leaking sprinkler heads will reduce *Phytophthora* problems.

**Seedling growth**—Growers routinely modify seedling development to meet certain physiological and mor-



Figure 33-5. Cover crops, such as sudan grass, are commonly grown in the fallow year between seedling crops in Pacific Northwest nurseries.



Figure 33-6. *Fusarium* growing on cover crop residue. Recent work at Oregon State University found higher soil populations of *Fusarium* and *Pythium* under cover crops than in fallow soil.

phological standards. These manipulations can also affect seedling health. Growers regularly adjust the sowing rate to compensate for seedlots that germinate poorly. It is tempting to oversow

(sow additional seed per square foot) to compensate for anticipated mortality from damping-off and *Fusarium* hypocotyl rot. But this is a questionable practice; disease is seldom uniformly distributed in



**Figure 33-7.** Water management is sometimes difficult in Pacific Northwest nurseries because of high rainfall and heavy soils. Tiling areas prior to sowing helps ensure good drainage.



**Figure 33-8.** Washing equipment helps to restrict movement of soilborne pathogens between blocks.

beds, and sowing too densely can result in increased losses to damping-off. The likely consequence of oversowing is poorly stocked patches of diseased seedlings interspersed among overstocked areas. High bed density creates a moist microclimate that favors stem

and foliage fungi and makes it difficult to get good fungicide coverage. Oversowing also wastes valuable seed and increases the likelihood that seedlings will be culled because they are too small. Compensating for disease losses can be done by sowing additional bed

feet at the same density, thus avoiding the problems of high seedbed densities.

Nitrogen fertilizers added early in the first growing season increase losses to damping-off and *Fusarium* hypocotyl rot. In soil of normal fertility, seedlings do not need supplemental nitrogen at this time.

Fertilization and irrigation are usually reduced to halt seedling growth before fall rains. Seedlings still growing into the fall are more vulnerable to upper stem canker and injury from early frosts. Root culture (pruning and wrenching) and top pruning are commonly practiced to curb and redirect seedling growth. These practices do not seem to affect pest development directly, although the clipped tops can support high gray mold populations.

*Sanitation*—Good nursery sanitation removes sources of possible infestation and checks the spread of many pests. Field equipment should be clean before it enters the nursery (Figure 33-8). It should be cleaned every time it is moved into a new field—particularly if into a fumigated field. Avoid moving transplant stock between or within nurseries since most soilborne fungi and insect pests can be spread this way.

Weed control is an important part of sanitation; weeds within the beds and in surrounding fields may harbor damaging insects. Similarly, hedgerow or windbreak trees may harbor pests that can damage nursery seedlings. Species for hedgerows and windbreaks should be selected to avoid those that are hosts for nursery seedling pests.

Seedlings with minor root infections are difficult to distinguish from healthy seedlings during grading. Mark infected areas before lifting and lift and destroy after crop seedlings have been harvested. Seedlings that are culled because of pest damage should be disposed of outside the nursery to prevent re-introduction of the pest into the nursery environment (Figure 33-9). Take care to avoid the contamination of healthy stock at lifting.



**Table 33-1. Commonly used fumigants for control of various soilborne diseases.**

| DISEASE                | FUMIGANT   |
|------------------------|--|
| Charcoal root disease  | methyl bromide + chloropicrin (67% + 33%)                        |
| Damping-off            | dazomet, methyl bromide + chloropicrin (67% + 33%), vorlex       |
| Fusarium hypocotyl rot | dazomet, methyl bromide + chloropicrin (67% + 33%)               |
| Fusarium root rot      | dazomet, methyl bromide + chloropicrin (67% + 33%)               |
| Phytophthora root rot  | dazomet, metam-sodium, methyl bromide + chloropicrin (67% + 33%) |

*Resistant seedlings*—Cultural control of some pests can also be obtained by growing seedlings that are less susceptible or even immune to damage. True fir species should not be grown in areas where *Phytophthora* has caused damage. Instead, sow or transplant resistant species such as western redcedar, pine, spruce, or larch. Likewise, if larch needle cast is a problem because of infected mature larch trees surrounding the nursery, consider contracting with another nursery with lower hazard to grow this species.

It is difficult to summarize the strategies for cultural control of nursery pests. Perhaps the best general idea to remember is that every nursery practice has a potential positive or negative impact on pests. Knowledge and careful use of cultural practices can help take advantage of the beneficial aspects and decrease the negative ones.

#### CHEMICAL CONTROL

Chemical control has been used for many years in forest nurseries. Pesticides can be classified into several categories depending on their target: soil fumigants, fungicides, nematicides, and insecticides. While specific products are mentioned in the following discussion, our purpose is not to make specific recommendations but rather to provide general information on why, how and when to use nursery pesticides.



**Figure 33-9. Cull seedlings should be disposed of well away from nursery beds.**

*Fumigants*—Soil fumigation is probably the most widely practiced chemical pest management technique in bareroot nurseries. Fumigation reduces losses to many soilborne diseases in the Pacific Northwest (Table 33-1). Fumigants kill weeds and weed seed, nematodes, soil insects, and pathogenic fungi. Crops grown in fumigated soil are regularly more vigorous and more uniform than crops from unfumigated beds. On the negative side, fumigants are expensive and extremely toxic to all living organisms. They are completely nonspecific in

their action and kill beneficial microorganisms in the soil as well as pathogens.

The most commonly used soil fumigants in the Pacific Northwest are dazomet and methyl bromide plus chloropicrin. Methyl bromide is a colorless and odorless gas. Chloropicrin is added and the mixture is injected in the soil and confined by a plastic tarp (Figure 33-10). Dazomet is a dry, micro-granular material that is incorporated into the soil by tilling or disking, and then sealed by rolling or watering the soil or placing a tarp over it (Figure 33-11).

**Table 33-2. Commonly used fungicides for control of various nursery diseases.**

| DISEASE                  | FUNGICIDE   |
|--------------------------|---|
| Damping-off              | benomyl, captan, ethazole, metalaxyl                    |
| Douglas-fir needle rust  | chlorothalonil, mancozeb, maneb                         |
| Fusarium hypocotyl rot   | benomyl, Banrot   |
| Fusarium root necrosis   | none  |
| Gray mold                | benomyl, captan, chlorothalonil, dichloran, vinclozolin |
| Larch needle cast        | benomyl, chlorothalonil, maneb                          |
| Lophodermium needle cast | chlorothalonil, mancozeb, maneb                         |
| Lower stem canker        | none  |
| Phoma blight             | chlorothalonil  |
| Phomopsis canker         | benomyl, chlorothalonil                                 |
| Phytophthora root rot    | metalaxyl   |
| Seed fungi               | captan, thiram  |
| Sirococcus tip blight    | chlorothalonil, triadimefon                             |
| Tip blight of pine       | chlorothalonil  |
| Upper stem canker        | benomyl, chlorothalonil                                 |
| Western gall rust        | none  |

The chemicals are most effective in warm and moist but not water-logged soils. Soils should be thoroughly moistened 10 to 14 days before fumigation to allow fungal propagules, weed seeds, and insect resting stages to imbibe water and begin to germinate or grow. These active stages are much more sensitive to the fumigant than the dormant stages.

The fumigant must be allowed to dissipate completely before a crop is sown. For this reason fumigation is usually done early in the fall of the year before sowing. If dazomet is applied in the spring, a bioassay must be done before planting to ensure that residual fumigant is no

longer present in the soil (Figure 33-12).

*Fungicides*—Several seedling diseases are readily controlled by fungicides. These chemicals are most often applied as foliar sprays but may also be used as seed treatments and soil drenches (Figure 33-13). Table 33-2 lists the most commonly used fungicides in the Pacific Northwest and the diseases they control.

Understanding fungicide action can help when planning treatments for specific diseases. Most fungicides must directly contact the target fungus or the target tissue to be effective. Foliar sprays should

coat exposed seedling parts with the chemical. If applied before infection occurs, they will kill germinating spores. Fungi that attack roots or stems beneath built-up soil collars are beyond the reach of contact fungicides. Some chemicals can also be applied as soil drenches, but their effectiveness is often reduced because they are inefficient in treating large volumes of soil. Most chemicals are inactivated rapidly in the soil by microbial action or are immobilized by binding to organic matter or clay particles.

Applying fungicides directly to seed before sowing is seldom a good strategy for protecting against pre-emergence damping-off and seed rot.

Many fungicides are toxic to germinating seeds and reduce the germination and growth of seedlings.

A few fungicides are absorbed by seedlings through roots or leaves and are translocated throughout the seedling. These systemic chemicals can stop established infections, and they often remain active longer than contact fungicides. In most cases, however, systemic fungicides cannot eradicate an established fungus; when treatment stops, the fungus may become active again.

The use of fungicides could be dramatically reduced in most nurseries in most years without any measurable effect on seedling health. The price of unnecessary chemical use includes risk of fungal tolerance to the fungicide, disruption of natural microbial balances, increased speed of environmental degradation, and adverse effects on human health.

Ironically, frequent use of some of the most effective chemicals, such as the systemic fungicides benomyl and metalaxyl, results in the development of tolerant strains of fungi. This is due to these chemicals' mode of action. In some nurseries where benomyl was used frequently and exclusively, tolerant strains of gray mold arose and predominated. Treatment that involves alternating among two or three different fungicides or mixing several fungicides in one application will delay the buildup of tolerant strains.

Here are some guidelines to help growers use fungicides properly:

1. Accurately diagnose the problem and determine if pest populations or environmental conditions are such that unacceptable damage will result.
2. Determine the most effective control measure, considering the use of a combination of methods (for example, chemical and non-chemical).
3. Use fungicides prudently, and only when other methods are inadequate.



**Figure 33-10.** Methyl bromide-chloropicrin fumigation is commonly used to decrease populations of nursery pests in the soil. Chemical is injected into the soil and then a tarp is laid to trap the gas.



**Figure 33-11.** Fumigation using dazomet. The microgranular material is tilled in and then the soil surface is sealed with water.



**Figure 33-12.** Fumigated soil should be tested for pesticide residue before sowing. Radish seed will not grow in soil containing residual fumigant.



**Figure 33-13.** Spraying pesticides on 1+0 seedlings is a common practice of disease control.

4. Select the appropriate fungicide, considering not only pest-control effectiveness but the effect of the fungicide on human health and the environment.

5. Use only fungicides registered for specific diseases on specific hosts.

6. Apply fungicides at the lowest possible dosage that will achieve disease control. Never exceed label rates.

7. Apply fungicides at the proper time and to the proper part of the seedling to control specific pathogens.

If fungicide treatment does not work as well as expected, one of several things may be wrong: the disease may have been misdiagnosed (perhaps it is caused by abiotic factors); there may be too much pathogen inoculum present; the fungicide may not be reaching the site of pathogen activity; the fungicide may have been applied too late, after infection had already occurred; resident pathogen populations may have acquired resistance to the chemical used.

*Insecticides*—Table 33-3 lists the insecticides commonly used in Pacific Northwest nurseries. Like fungicides, they are probably applied more frequently than is necessary. The most important aspect of successful use is timing the application to coincide with a vulnerable life stage of the insect. Nurseries with good monitoring programs generally prevent insect damage most successfully, and do so with fewer applications of chemicals. On the other hand, if insecticides are not used until widespread crop damage is apparent, the opportunity to halt the epidemic with insecticides has usually passed. This is particularly true with larvae that feed beneath the soil; they must be controlled with sprays timed to catch the adults in their flight period, before they lay eggs. Sometimes sprays can be fine-tuned to the diurnal activity of the

**Table 33-3. Commonly used insecticides for control of various nursery insect pests.**

| INSECTS                    | INSECTICIDES   |
|----------------------------|--|
| Cooley spruce gall adelgid | carbaryl, endosulfan   |
| Cranberry girdler          | chlorpyrifos, diazinon   |
| Cutworms                   | diazinon, esfenvalerate <sup>1</sup>                           |
| Grasshopper                | carbaryl, diazinon   |
| June beetle (white grubs)  | dazomet, methyl bromide + chloropicrin (67% + 33%) (fumigants) |
| Leafrollers                | acephate, diazinon   |
| Lygus bugs                 | acephate, esfenvalerate <sup>1</sup>                           |
| Pine bark aphid            | endosulfan   |
| Root weevils               | bendiocarb, orthene  |
| Seedcorn maggot            | chlorpyrifos, diazinon   |

<sup>1</sup>The insecticide fenvalerate has been replaced by esfenvalerate, a chemical with similar properties.

target pest.

Insecticide efficacy can be improved by continuing to monitor beds for insects and damage after treatment. The presence of target insects or seedling damage despite treatment may indicate the use of the wrong insecticide or improper timing or mixing of insecticides. Some nurseries have designated small check areas to evaluate insecticide applications. Periodic reviews of application records and data on pest damage are also useful.

Seedlings can be damaged if insecticides are improperly mixed or applied. Germinants and succulent 1+0 stock are particularly susceptible to phytotoxicity. Spraying in the cool of the morning and using wettable powders instead of emulsifiable concentrates can reduce the risk to sensitive stock. Tank mixing of insecticides with other chemicals should be done cautiously because of the potential for phytotoxic effects.

*Nematicides*—*Nematodes* seldom cause measurable losses in Pacific Northwest nurseries because the soil is regularly fumigated. Nematicides are therefore rarely used. Nematode populations, however, can increase and cause losses in transplant beds, particularly 1+1 or plug+1, if the beds have gone several years without fumigation. Soil nematode assays and regular monitoring of damage can identify potential trouble spots. The nematicides fenamphos, oxamyl, and dichloropropene are registered for use. The first two chemicals, which are applied after planting, have been tested and found effective for controlling nematodes on conifers in the Pacific Northwest. Dichloropropene is a soil fumigant used before sowing. It is intended primarily to control nematodes, but when chloropicrin is added it affords some control over soilborne fungi as well.

#### BIOLOGICAL CONTROL

Biological control consists of a relatively new and mostly untried set of strategies for controlling plant pests in forest nurseries. A biological control organism generally is a naturally occurring insect, fungus, or bacterium that either harms plant pests or benefits the host.

Predatory insects, as the name implies, lower the pest insect population directly by feeding on the pest. An example of this kind of biological control organism—one that nurseries have been using for some time—is ladybugs that feed on adelgids. Some fungi and bacteria produce toxins or antibiotics that inhibit growth of pathogenic organisms. Other beneficial microorganisms produce chemicals that increase seedling growth and thus reduce pest damage. Several such biological control agents are currently being investigated in the hope of using them to provide protection in the future.

Biological control has at least two potential advantages over chemical

control. First, a biological control organism may grow and reproduce on the seedling; the population of a single application may protect the plant throughout the period of susceptibility. This contrasts with the need to spray certain pesticides several times to obtain good disease control. Secondly, most biological agents pose little or no threat to workers or the environment. Thus they can safely be applied several times if necessary.

Developing a biological control agent for bareroot nurseries is a difficult undertaking at best. The organism used has to be able to survive and grow in competition with all the many other organisms found on seedlings and in nursery soil. Thus, it is more difficult to develop a biological control that would be useful in outdoor, field situations than it is to develop one to control pests in indoor container nurseries.

## Pest management checklist

The following checklist can help nurseries reduce losses from pests. If nursery managers monitor effectively, keep good records, and follow this checklist, they should be able to achieve a comprehensive, cost-effective pest control program without compromising the environment, the safety of the workplace, or the quality and quantity of seedlings.

### BEFORE SOWING

#### *Soil management*

- Are drainage problems corrected?
- Is a cover crop necessary?
- Are organic amendments necessary?
- If sawdust is added, is it free of pests and other contaminants?
- Is supplemental nitrogen needed to grow cover crop and to break down organic matter?
- Are beds raised in preparation for sowing?

#### *Fumigation*

- Is fumigation necessary?
- Should it be done in fall or spring?
- Which chemical should be used?

Has adequate moisture been maintained 10-14 days before fumigation?

Are soil moisture and temperature favorable?

#### *Pest monitoring*

Have pre- and post-fumigation *Fusarium* assays been done to test effectiveness of fumigation?

Has an assay for seed fungi been done in selected seedlots?

#### *Seed*

Have precautions been taken to assure clean seed?

Have special protective measures been taken for seedlots with low vigor or high fungal populations?

#### *Sanitation*

Is equipment cleaned before working fumigated beds?

Is a clean water supply assured?

Have sources of pathogenic fungi in surrounding windbreaks or cull piles been removed?

### 1+0 YEAR

#### *Pest monitoring*

Have damage surveys been taken at appropriate times for damping-off, *Fusarium* hypocotyl rot and root rot, cutworms, and upper stem canker?

Has timely pest monitoring been done for lygus bugs and other insects and diseases of 1+0 seedlings?

#### *Direct control*

Based on history and current monitoring results, is pesticide application necessary for damping-off, insect pests, or upper stem canker?

#### *Cultural practices*

Have irrigation requirements been coordinated for growth, cooling, and frost protection to avoid excessive water use?

Are fertilization and irrigation coordinated to stop seedling growth before fall rains?

Is mulching necessary to protect against lower stem canker and *Phoma* blight?

Is ripping necessary to improve winter drainage?

#### *Sanitation*

Is equipment regularly washed as it is moved between fields?

Are weeds controlled to deny refuge to damaging insects?

### 2+0 YEAR

#### *Pest monitoring*

Are timely damage surveys planned for *Phytophthora* root rot and gray mold?

Are timely pest surveys planned for lygus bugs and cranberry girdler moths?

#### *Direct control*

Based on history and current monitoring results, is pesticide use warranted for control of *Phytophthora*, lygus bugs, gray mold, or cranberry girdlers?

#### *Cultural practices*

Have irrigation requirements been coordinated for growth, cooling, and frost protection to avoid excessive water use?

Are fertilization and irrigation coordinated to stop seedling growth before fall rains?

Is ripping necessary to improve winter drainage?

Will top pruning cause gray mold problems?

#### *Sanitation*

Is equipment regularly washed as it is moved between fields?

Are weeds controlled to deny refuge to damaging insects?

### TRANSPLANTS

#### *Pest monitoring*

Has transplant stock been checked for pests?

Is *Phytophthora* a likely problem?

Is a nematode assay warranted?

*Direct control*

Based on nursery history, monitoring, and source of transplants, is pesticide application necessary to control *Phytophthora*, nematodes, or other pests?

*Cultural practices*

Is drainage corrected before transplanting?  
Are beds raised?  
Has care been taken to avoid overwatering or underwatering while transplants are getting established?

*Sanitation*

Is equipment regularly washed as it is moved between fields?  
Are weeds controlled to deny refuge to damaging insects?

## LIFTING AND STORAGE

*Pest monitoring*

Have seedlings with *Phytophthora* infection been identified?  
Has gray mold been controlled prior to lifting?  
Are storage containers monitored for mold development?

*Direct control*

Are fungicide treatments warranted before seedlings go into storage?

*Cultural practices*

Are plans made to keep seedlings clean, cool, and moist during lifting and sorting?  
Do storage facilities have an adequate temperature monitoring, alarm, and recording system?  
Are cooling facilities adequate to promptly establish and maintain target temperatures inside storage containers?

*Sanitation*

Are *Phytophthora*-infected seedlings lifted, sorted, and stored separately from healthy trees?

Are culls safely disposed of outside the nursery?  
Is seedling foliage generally free of soil going into storage?  
Are moldy seedlings removed from storage and disposed of, or outplanted as soon as possible if mold is superficial?

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# Glossary

**Abiotic damage**

Damage to plants caused by non-living agents such as heat, frost, or fertilizers.

**Amendment, soil**

Any substance added to soil to alter its physical or chemical properties and thereby make it more useful for plant production.

**Asexual spore**

A spore produced by mitosis; in contrast to a sexual spore produced by meiosis.

**Attractant, insect**

A substance that lures insects to traps or poison-bait stations. Different types are usually classified as food, oviposition, and sex attractants.

**Biological control**

The use of parasites, predators, or other living organisms to suppress pest populations or prevent pest damage.

**Blight**

Common name for a number of different diseases on plants, especially when plant tissue injury occurs suddenly; for example, needle blight, blossom blight, and shoot blight.

**Bulk density**

*The weight per unit* volume of a soil, including both solid particles and air spaces.

**Calcareous**

Soils with a high calcium content. Calcareous soils often have a high pH.

**Cambium**

In woody plants, the thin layer of cells between the xylem and phloem that gives rise to new cells.

**Canker**

A killed area on the stem or branch of a plant, usually shrunken and oval or circular in shape.

**Cation exchange capacity**

A measure of the ease with which cations (positively charged ions such as  $\text{Ca}^{2+}$  or  $\text{K}^{+}$ ) are held on negatively charged sites on clay or humus particles.

**Causal organism**

The pathogen that causes a given disease.

**Chlamydospores**

Thick-walled fungus spores produced asexually. Often important for survival of the fungus during unfavorable conditions.

**Chlorosis**

Yellowing of foliage from loss of chlorophyll. Can be caused by a variety of biotic and abiotic factors.

**Conidia**

Asexual reproductive spores of fungi, often produced in great numbers. Also called conidiospores.

**Cortex**

Tissues of a young seedling stem or root lying between the vascular tissues and the epidermis.

**Cotyledons**

The seed leaves of a plant. In conifers, the cotyledons are first to

emerge and carry the seed out of the soil.

**Cover crops**

Crops grown principally to control various forms of erosion but also incorporated into the soil to increase organic matter. Grown in rotation with seedlings.

**Crozier**

A shepherd's crook; refers to crook in seedling stem.

**Cull**

A seedling that is not acceptable because it does not meet certain size and quality standards. Culls are thought to have low outplanting survival and growth potential.

**Cultural control**

The use of certain nursery practices (for example, controlling weeds, improving drainage, or adding soil amendments) to make the habitat less favorable for pests or to prevent, suppress, or remove them.

**Cuticle**

Waxy layer on the outside of a leaf.

**Ectoparasite**

A parasite that feeds on the host from outside the plant.

**Epidermis**

The layer of cells just beneath the cuticle on a stem or leaf.

**Fallow**

To allow cultivated land to remain idle during most or all of the

growing season, usually as a crop rotation technique between seedling crops.

### **Fertilizer burn**

Chlorosis or necrosis of seedling tissue resulting from excessive or misapplied fertilizer.

### **Fibrous root system**

A desirable root form that contains a mass of fine roots.

### **Fumigant**

A soil-applied chemical that volatilizes to a gas and is used to kill insects, fungi, nematodes, or bacteria, as well as seeds, roots, rhizomes, or entire plants.

### **Fungicide**

A chemical used to kill or inhibit fungi.

### **Germ tubes**

The hyphae that first emerge from spores.

### **Holdfast**

Specialized fungus cell that attaches to the surface of the host.

### **Host**

The organism that is attacked, infected, or otherwise damaged by a pest.

### **Hibernaculum**

Chamber made from "silken" web in which insect larvae hibernate.

### **Hyphae**

A single vegetative filament of a fungus.

### **Hypo cotyl**

The portion of a seedling between the cotyledons and the root.

### **Inoculation**

The transfer of a pathogen onto a host.

### **Inoculum**

That part of a pathogen that causes initial infection of a host; a spore, for example.

### **Insecticide**

A chemical used to kill or inhibit insects.

### **Instar**

The stage of development that occurs between molts of the larvae of an insect.

### **Integument**

The inner layer of the seed coat.

### **Lateral branch**

Side branch of a seedling.

### **Lesion**

A localized area of dead tissue on a root, stem, or leaf.

### **Macroconidia**

The larger of two kinds of asexual spores produced by fungi such as *Fusarium*.

### **Meiosis**

Division of nuclei that reduces the chromosome number by half and rearranges the genes; in contrast to mitosis, in which nuclei are copied exactly without change in chromosome number or arrangement.

### **Microsclerotium**

Small, thick-walled, multi-celled resting structure produced by some fungi.

### **Monoclonal antibody**

A chemical molecule, produced in the immune response of an animal, that "recognizes" a specific protein. Used to detect and identify specific pathogens.

### **Multiseptate**

Having several septations, or crosswalls.

### **Mycelium**

The collective mass of vegetative filaments, or hyphae, of a fungus.

### **Mycorrhizae**

The symbiotic association between plant roots and particular fungi.

### **Necrosis**

Death of plant cells or tissues.

### **Nymphs, insect**

Immature adult insects; their form resembles that of the adult.

### **Oospore**

Sexual spore produced by the water molds. Commonly acts as a resting spore when soil conditions are unfavorable for fungus growth.

### **Oviposit**

To lay eggs.

### **Pathogen**

Specific agent that can cause disease. Usually a fungus, bacterium, virus, or nematode.

### **Pathogenicity**

The capacity of an organism to cause disease.

### **Perfect stage**

That portion of the life cycle of a fungus in which sexual fusion and meiosis take place.

### **Pesticide**

Any substance used to kill or inhibit any pest. Includes fungicides, herbicides, fumigants, insecticides, nematocides, rodenticides, dessicants, defoliants, plant growth regulators, and others.

### **pH**

Numerical measure of the acidity (<7) or alkalinity (>7) in a soil or solution. A pH reading of 7 is neutral.

### **Phloem**

Portion of the vascular system of a seedling that is responsible for the downward transportation of sugars from the needles to the roots. Formed just outside the cambium, the phloem is also called the inner bark.

### **Preemergence**

The time period after sowing and before crop plants emerge.

**Primary inoculum**

Inoculum that causes the first infections in the crop; usually inoculum produced by overwintering structures such as chlamydospores or sclerotia.

**Propagule**

A reproductive unit of the pathogen; spores, hyphae, or microscletia of fungi, for example.

**Pupa**

Insect developmental stage between larvae and adult. Often a resting stage.

**Pycnidia**

Flask-shaped fruiting body of a fungus; produces conidia.

**Sanitation**

Removal of infested or infected plants or plant parts from the growing site to prevent spread of the pest to healthy plants.

**Saprophyte**

An organism that lives on dead organic matter.

**Sclerotium**

Thick-walled, multiple-celled resting structure of a fungus.

**Secondary inoculum**

Inoculum that is produced on the plant as the result of earlier infection.

**Soil texture**

The proportion of coarse and fine particles in a soil.

**Sporangium**

A fungus cell that holds asexual reproductive spores, often zoospores.

**Spore**

A single- to many-celled reproductive body in fungi that can develop into a new fungus colony.

**Sporodochium**

Mound-shaped asexual fruiting body of a fungus.

**Stomate**

Pore in the leaf used for gas exchange in transpiration and photosynthesis.

**Straw dust**

A mulching material made primarily from ground-up grass straw.

**Stylet**

An elongated piercing mouthpart of an insect or nematode.

**Symptom**

The evidence of disease or injury, such as wilting, yellowing, or death of tissues.

**Systemic**

Entering and then acting within the entire organism. Used especially to describe the action of pesticides or diseases within a plant.

**Terminal**

The uppermost shoot or leader of a seedling.

**Witches' broom**

An abnormal proliferation of lateral branches on a stem.

**Xylem**

Portion of the vascular system of a seedling responsible for the upward transportation of water and nutrients from the roots to the stem and leaves. Formed just inside the cambium.

**Zonal pattern**

Pattern of root development where rooting depth is limited by a layer of compacted soil.

**Zoospore**

Asexual reproductive spore that swims in water. Produced by the water molds such as *Pythium* and *Phytophthora*.

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