Nematode Damage and Management in North American Forest Nurseries

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Abstract

Plant-parasitic nematodes can affect seedling production in forest nurseries when host seedlings are developmentally vulnerable, nematode populations are high, or opportunistic pathogens are present. Soil fumigation has been important for plant-parasitic nematode control in forest nurseries. Regulatory changes and rising costs of fumigant application are expected to affect nursery pest management programs. In the future, management strategies for the control of various nematodes will increasingly depend on the biology of the nematodes and hosts. This article provides a brief review of nematode problems that affect seedlings in forest nurseries, symptoms of nematode damage, and nematode control practices.

Introduction

Nematodes have long been associated with bareroot seedling damage in North American forest nurseries and some plant-parasitic species cause significant stunting and chlorosis of seedlings (Hopper 1958, Johnson and others 1970, Sutherland and Sluggett 1975, Peterson and Riffle 1986, Fraedrich and Cram 2002). Plant-parasitic nematodes are microscopic worms that feed on plants by removing the cell contents with a hollow, needle-like mouthpart called a stylet, which functions much like a straw (figures 1 and 2). Some plant-parasitic nematodes remain in the soil and feed by repeatedly thrusting their stylets into seedling roots. These nematodes are referred to as ectoparasites. Other plant-parasitic nematodes are endoparasites and invade root systems to feed inside the root tissues. Among the numerous species of plant-parasitic nematodes, many are specialized to attack various types of plant tissues including leaves, flowers, stems, and roots; however, most damaging nematodes are soilborne and feed on roots (Shurtleff and Averre 2000). In some instances, root diseases can develop from the interaction of the physical damage caused by nematode feeding and soilborne fungal pathogens that colonize the wounded root tissues (Dwinell and Sinclair 1967, Shurtleff and Averre 2000).

The environmental conditions in most forest nurseries are ideal for many species of plant-parasitic nematodes. In addition to high host densities, bareroot nurseries are typically located on well-drained sandy soils that are irrigated regularly. Highly porous soils, where pore sizes exceed 30 microns, allow for the free movement of most plant-parasitic nematodes. Large

Figure 1. A translucent, worm-shaped stunt nematode (*Tylenchorhynchus ewingi*.) (Photo source: Stephen W. Fraedrich).

Figure 2. Head of a *Tylenchorhynchus ewingi* nematode with a clearly visible stylet. (Photo source: Michelle M. Cram).

nematodes such as *Xiphinema* spp. and *Longidorus* spp. require larger pore sizes (60 microns) and are typically found in coarse-textured, sandy soils (Jones and others 1969, Norton 1979). Although these soils dry quickly, which could immobilize and desiccate nematodes, nursery irrigation for optimum seedling growth also provides optimal conditions for nematode movement and survival (Jones and others 1969, Norton 1979).

Prior to the 1970s, cases of severe seedling losses thought to be associated with nematodes were controlled in research studies by a variety of soil fumigants (Henry 1953, Bloomberg and Orchard 1969, Peterson 1970). Eventually, methyl bromide fumigation became the standard preplant soil treatment in nurseries (Fraedrich and Dwinell 2005, Zasada and others 2010). In a 1993 national survey of forest nursery managers, soil fumigation for nematode control had some importance to 44 of 52 southern nurseries, 28 of 35 northeastern nurseries, and 10 of 21 western nurseries (Fraedrich 1993).

Although fumigation is initially highly effective for reducing nematode populations, the populations will rebound over the growing season and can damage the next seedling crop unless preplant control practices are applied again (Fraedrich and others 2003, Fraedrich and Dwinell 2005, Enebak and others 2011). An integrated pest management program that uses fumigants in conjunction with periods of fallow and rotations with nonhost cover crops is expected to provide increased control of plant-parasitic nematodes and associated diseases (Fraedrich and others 2005).

Plant-Pathogenic Nematodes of Forest Nurseries

Surveys of forest nurseries in North America before 1970 documented numerous species of plant-parasitic nematodes in the soils; however, only a few species were thought to be associated with seedling injury (Hopper 1958, Peterson 1962, Sutherland and Dunn 1970). Nematode species currently known to damage seedlings in forest nurseries are listed in table 1. This list is likely incomplete because many more nematode species are capable of feeding on roots of forest

Table 1. Plant-parasitic nematodes known to damage seedlings in the North America forest nurseries.

Feeding class	Common name	Nematode species	Tree host(s)	Citation
Endoparasite Migratory	Lance	*Hoplolaimus cronatus* Cobb	*Pinus taeda* L., *Pinus elliottii* Engelm. var. *elliottii*	Ruehle 1962, Ruehle and Sasser 1962
	Root-lesion	*Pratylenchus brachyurus* (Godfrey) Filip. & Stek.	*Pinus palustris* Mill., *Pinus taeda*	Hopper 1958, Ruehle 1973a
		P. penetrans (Cobb) Chitwood and Oteifa	*Pseudotsuga menziesii* (Mirb.) Fran	McElroy 1985
			Juniperus virginiana L., *Pinus ponderosa* P. & C. Lawson	Caveness 1957, Peterson 1970, Viglierchio 1979
Sedentary	Pine cystoid	*Meloidodera floridensis* Chitwood, Hannon, and Esser	*Pinus taeda, Pinus elliottii* var. *elliottii, Pinus clausa* (Chapm. ex Engelm.) Vasey ex Sarg.	Hopper 1958, Ruehle and Sasser 1962, Ruehle 1973a
	Root-knot	*Meloidogyne javanica* (Treub) Chitwood	*Pinus elliottii* var. *elliottii Liriodendron tulipifera* L.	Donaldson 1967, Ruehle 1971
		M. incognita (Kofoid and White) Chitwood	*Cornus florida* L.	Johnson and others 1970
Ectoparasite	Sting	*Belonolaimus* sp.	*Pinus* sp.	Esser 1977
	Needle	*Longidorus americanus* Handoo	*Pinus taeda, Pinus elliottii* var. *elliottii*	Fraedrich and Cram 2002, Fraedrich and others 2003
			Quercus spp.	Fraedrich and others 2003, Cram and Fraedrich 2005
	Stunt	*Tylenchorhynchus claytoni* Steiner	*Pinus palustris, Pinus taeda, Pinus elliottii* var. *elliottii*	Hopper 1958, Ruehle 1973a, Cram and Fraedrich 2009
		T. ewingi Hopper	*Pinus elliottii,* var. *elliottii* *Pinus taeda*	Hopper 1959, Fraedrich and others 2012
	Dagger	*Xiphinema bakeri* Williams	*Pseudotsuga menziesii Tsuga heterophylla* (Raf.) Sarg. *Picea stchensis* (Bong.) Carr. *Picea glauca* (Moench) Voss	Sutherland 1970, Bloomberg and Sutherland 1971, Sutherland and Sluggett 1975
	Stubby-root	*Paratrichodorus minor* (Colbran) Siddiqi	*Pinus elliottii* var. *elliottii, Pinus taeda, Pinus palustris*	Ruehle 1969
	Spiral	*Helicotylenchus nannus* Steiner	*Pinus taeda*	Ruehle and Sasser 1962

seedlings, but their potential to cause significant damage and affect seedling growth has not been fully investigated. Likewise, various plant-parasitic nematode species are associated with outplanted seedlings or mature trees (Ruehle and Sasser 1962, Ruehle 1966, Riffle and Kuntz 1967, Sanzo and Rohde 1967, Ruehle 1968b, Riffle 1970, Churchill and Ruehle 1971, Riffle 1972, Ruehle 1972, Ruehle 1973b, Maggenti and Viglierchio 1975, Viglierchio 1979, Eisenback and others 1985), but these parasitic species are not listed in table 1 because they have not been documented to cause problems on seedlings in forest nurseries.

The level of damage caused by plant-parasitic nematodes is often determined by the age of seedlings when the first attack occurs and the population densities of nematodes in the fields. Seedlings are most susceptible to the damage caused by plant-parasitic nematodes during the weeks after seed germination. Studies using agricultural crop plants have shown that delaying nematode attacks on young seedlings by several weeks can dramatically reduce nematode effects on plant growth and final development (Wong and Mai 1973, Seinhorst 1995, Ploeg and Phillips 2001). Nematode host studies in forestry have often used seedlings 2 to 9 months old or low population densities that are only adequate to determine if a nematode species was parasitic on a crop. Older seedlings can better tolerate the effects of some nematode feeding without losses in seedling quality, similar to how they can tolerate the effects of undercutting and lateral pruning. Controlled studies that applied nematodes to soil before sowing or at the time of seed germination have typically shown significant seedling growth losses (Ruehle 1969, Sutherland and Sluggett 1975, Fraedrich and others 2003, Cram and Fraedrich 2009, Fraedrich and others 2012). Other studies have demonstrated significant seedling growth losses by using high population densities or by assessing damage over longer periods of time (Ruehle and Sasser 1962, Ruehle 1968a, Johnson and others 1970, Ruehle 1973a). High populations of plant-parasitic nematodes in forest nurseries that occur later in the growing season will likely cause some reduction in seedling growth. The degree of stunting, however, will not be as devastating as when young seedlings are attacked early in the growing season.

Few studies have attempted to determine relationships between nematode population densities before seed sowing and subsequent damage to forest-tree seedlings. The number of nematodes associated with damaged bareroot seedlings varies by nematode species, host species, and timeframe. Many assessments of nematode populations have been made only when damaged seedlings have been observed. For example, 315 to 422 *Tylenchorhynchus claytoni* Steiner per 100 cc soil were associated with severe injury of *Pinus* seedlings (Hopper 1958), and 40 to 208 *Longidorus americanus* Handoo per 100 g soil were associated with stunted loblolly pine (*Pinus taeda* L.) seedlings (Fraedrich and Cram 2002). These field cases are of little value for predicting the potential for seedling damage at the beginning of a growing season. Controlled studies that examined the effect of a range of nematode population densities on young seedlings have shown that much lower populations can damage seedlings. Several tests with stunt nematodes demonstrated that 60 nematodes per 100 cc soil, at or within 1 month of germination, could significantly reduce root weight of seedlings (Ruehle 1973a, Fraedrich and others 2012). The much larger nematode, *L. americanus*, decreases seedling root weights if only 30 nematodes per 100 g soil are present in soil at the time of seed germination (Fraedrich and Cram 2002, Fraedrich and others 2003). More research is needed on individual nematode pests and their effect on tree seedlings to better determine the relationship between population density and economic losses.

Plant-parasitic nematodes can become established in forest nurseries in several ways. In many cases, the nematodes were probably already established in fields when the land was converted from agricultural crop production or forests. Nursery fields can also become infested with nematodes by soil movement through mechanical means (e.g., tractors, equipment), wind or flooding, and by transplanting infected plants (Shurtleff and Averre 2000). Sutherland and Dunn (1970) found greater populations of *Xiphinema bakeri* Williams in British Columbia where field soils were ameliorated with sand to increase the porosity. A similar case was documented in a Florida nursery where a *Belonolaimus* sp. was brought in with forest soil that was used to fill a low area (Esser 1977). In the case of *L. americanus* at a Georgia nursery, we speculate that the nematode was introduced to fields during a flood, which occurs periodically because of the nursery's close proximity to a major river. After a plant-parasitic nematode is introduced into a nursery, it is unlikely to be eradicated and therefore will require a long-term management plan.

Although it is possible that plant-parasitic nematodes are transported to outplanting sites, there is no documented case of nematodes from a nursery affecting outplanted seedlings. Ruehle and Sasser (1962) attempted to investigate whether nematodes from a North Carolina nursery were the cause of stunting in outplanted seedlings. They found that pine cystoid nematodes indigenous to the outplanting site were the cause of damage, while the lance nematodes from the planting stock were nearly nonexistent after 2 years.

Symptoms of Damage by Plant-Parasitic Nematode

The aboveground symptoms associated with nematode damage can be highly variable. In some cases, the seedlings will be severely stunted, chlorotic, and even wilted (figure 3). In other cases, the symptoms caused by nematodes may be much less severe and primarily noted because seedlings are growing slower than normal and are off color. Adequate moisture and fertilizer can sometimes compensate for nematode feeding and minimize aboveground symptoms (Ruehle 1973b). In some cases, symptoms of nematode damage may be confused with other factors, including nutrient deficiencies, root disease, insect damage, seasonal effects, and inadequate or excess water (Ruehle 1973b, Shurtleff and Averre 2000). These factors sometimes occur in combination with nematode damage, thereby complicating identification of the primary cause of damage. Nematode injury can predispose seedling roots to opportunistic and pathogenic fungi resulting in greater damage and root rot (Bloomberg and Sutherland 1971, Ruehle 1973b, Barham and others 1974). The ability of roots to form mycorrhizae is also impeded by nematodes (Ruehle 1973b). Ultimately, nematode-damaged seedlings with compromised root systems can have difficulty absorbing water and nutrients, which can be misdiagnosed as a nutrient-deficiency problem.

The distribution of damage in nursery fields can be somewhat helpful in the diagnosis of nematode problems. Early in an infestation, the pattern of damage often occurs as discreet patches of affected seedlings, which can expand to larger areas that encompass entire fields (figures 4 and 5). Nursery equipment will move soil and nematodes within a field and to other uninfested fields. A recent example of this contamination occurred in a Georgia nursery where *L. americanus* initially caused seedling stunting in a few small patches of 3 to 9 m (6 to 27 ft) of nursery bed that within a few years spread throughout one-half of a 10-acre (4-hectare) field (Fraedrich and others 2003).

The feeding class of a plant-parasitic nematode will often affect the type of symptoms observed on roots. Migratory endoparasitic nematodes colonize roots and frequently cause necrotic lesions that allow bacteria and fungi to colonize. Other endoparasitic nematodes become sedentary and stimulate the formation of root galls or swellings. *Meloidogyne* spp. (root-knot nematodes) are known to form galls on hardwoods, but only cause a slight root swelling in conifers (Ruehle 1973b). *Meloidodera* spp. (cystoids nematodes) also produce only a slight swelling on pines and, at maturity, their bodies can protrude from roots and appear much like small pearls on the surface of roots (Ruehle and Sasser 1962). Some ectoparasitic

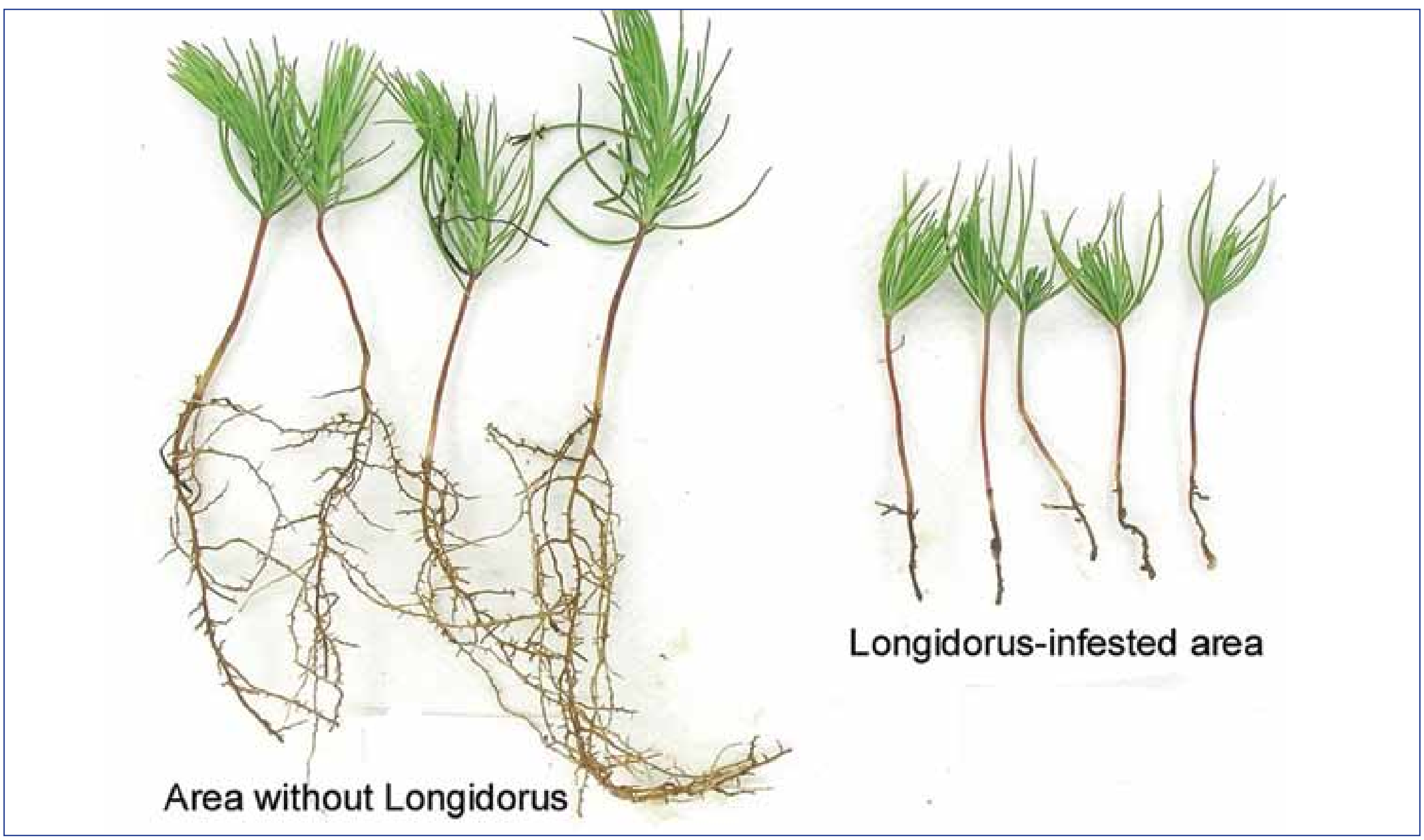

Figure 3. Loblolly pine (*Pinus taeda*) seedlings from nursery beds infested and uninfested by *Longidorus americanus*. (Photo source: Stephen W. Fraedrich).

Figure 4. Patches of stunted loblolly pine (*Pinus taeda*) seedlings damaged by *Longidorus americanus*. (Photo source: Stephen W. Fraedrich).

Figure 5. Chlorotic and stunted slash pine (*Pinus elliottii* Engelm. var. *elliottii*) seedlings damaged by *Tylenchorhynchus claytoni*. (Photo source: Michelle M. Cram).

nematodes that feed near the root tip, such as *Longidorus* and *Xiphinema* spp., can also stimulate swellings consisting of compact parenchyma cells (Ruehle 1973b). Feeding by most ectoparasitic nematodes, however, results in suppressed cell division and reduced water and nutrient uptake (Shurtleff and Averre 2000). Roots become underdeveloped and stubby when fed upon by ectoparasitic nematodes such as *Tylenchorhynchus* spp. and *Paratrichodorus* spp.

Collecting Samples

To determine if plant-parasitic nematodes are a problem or have the potential to become a problem in nursery crops, soil samples need to be sent to laboratories that offer nematode identification services. The use of a number of nematode extraction techniques may be necessary to diagnose nematode problems. Techniques used to diagnose problems caused by sedentary, endoparasitic nematodes often differ from those caused by ectoparasitic nematodes. It may also be necessary to request that the laboratory use techniques specifically for larger nematodes, such as *Longidorus* spp., as well as standard techniques used for quantification of smaller nematodes like *Tylenchorrhynchus* spp. The identification of *L. americanus* as the cause of severe stunting of loblolly pine seedlings in a Georgia nursery was delayed due to the testing laboratory's use of a sugar floatation method that is better suited for smaller nematodes (Fraedrich and Cram 2002). Extraction techniques for larger nematodes require some minor modifications of standard techniques used for smaller nematodes (Flegg 1967, Shurtleff and Averre 2000, Fraedrich and Cram 2002).

Soil samples for nematode extraction need to always be taken in the root zone, generally in the upper 15 to 20 cm (6 to 8 in). If a nursery manager is assessing a field before growing seedlings, a composite soil sample should be obtained that consists of 20 to 25 samples from across the field (Shurtleff and Averre 2000). For larger fields and for more accurate information about the risks of particular nematodes, the field needs to be divided and sampled by quadrant. If a problem is being diagnosed during a growing season, a composite sample consisting of several subsamples needs to be taken from the root zone of affected seedlings. Samples of seedlings should also be taken and sent with the soil samples. The best location to sample for plant-parasitic nematodes is normally towards the edges of patches of stunted seedlings. Avoid sampling soil and roots of severely damaged seedlings in the center of stunted seedling patches because the nematode populations have usually declined and moved outward to the patch edges where seedlings have larger root systems (Shurtleff and Averre 2000, Fraedrich and Cram 2002). The moisture level of soil needs to be neither excessively wet nor dry at the time of sampling. Sampling when moisture levels are between 75 to 100 percent of field capacity is best for nematode survival (Norton 1979). Nematodes are essentially aquatic worms that require water to survive and move in soils; therefore, samples need to always be placed in plastic bags to maintain the moisture level. Prevent samples from being exposed to temperatures less than 4 °C (40 °F) or greater than 27 °C (80 °F). Nematodes can be stored in plastic bags for a few weeks at temperatures between 4 °C and 18 °C (40 °F to 65 °F) (Shurtleff and Averre 2000).

Control of Plant-Parasitic Nematodes

Forest nurseries routinely practice an integrated approach to manage most soilborne pests, including plant-parasitic nematodes. The average nursery fumigates fields and then produces seedlings for 2 years followed by 1 or 2 years of green-manure crops. This combination of fumigation with crop rotation can help to reduce many soilborne pests. When a nematode problem does occur, most nurseries use sanitation measures to avoid infesting new fields. Ultimately, managers need to know what species of plant-parasitic nematodes are present and the host range of those nematodes to develop an effective management strategy.

Soil fumigation has been the primary means of controlling plant-parasitic nematodes in forest nurseries for the past four decades (Fraedrich and Dwinell 2005, Zasada and others 2010). Prior to the 1970s, seedling losses due to damage associated with nematodes were routinely reported by nurseries throughout North America (Hopper 1958, Johnson and others 1970, Sutherland and Sluggett 1975, Peterson and Riffle 1986). Early trials of fumigants for forest nurseries found that chloropicrin, methyl bromide, and methylisothiocyanate products such as Vapam significantly reduced nematode populations and improved seedling growth (Henry 1953, Hansbrough and Hollis 1957, Bloomberg and Orchard 1969). By the late 1980s, methyl bromide with chloropicrin was the primary fumigant for many growers in the United States who relied on preplant fumigation for their crops (Zasada and others 2010). This combination remains the preferred fumigant for forest nurseries in some parts of the United States to this day, despite the continued phase out of methyl bromide under the Montreal Protocol and Clean Air Act (Enebak and others 2011).

Research conducted in forest nurseries during the past two decades to find replacement fumigants for methyl bromide has found that most alternative fumigants provide good control of plant-parasitic nematodes (Fraedrich and Dwinell 2005, Cram and others 2007, Enebak and others 2011). Although fumigants are highly effective against nematodes, fumigation does not eradicate nematodes in fields (Lembright 1990) because toxic concentrations of the fumigants may not reach all plant-parasitic nematodes throughout the soil horizon (McKenry and Thomason 1976, Lembright 1990). Nematodes may survive fumigation if they are located beneath the fumigant's effective concentration zone or if they occur in areas of the soil where moisture levels are too high for effective fumigation. Nematodes may also occur in soil clods or hardpans where the fumigant is excluded. Endoparasitic nematodes can escape if the fumigant fails to penetrate the host's roots. A fumigant may also be ineffective when nematodes are in a more tolerant form such as a cyst or an anhydrobiotic state. Nematode population densities often begin to rebound during the first year and can reach sufficiently high levels to damage subsequent seedling crops (McKenry and Thomason 1976, Fraedrich and Dwinell 2005, Enebak and others 2011). Populations of plant-parasitic nematodes can increase very rapidly in fields because of their relatively short life cycles (3 to 6 weeks), plentiful egg production, and abundance of host roots (Shurtleff and Averre 2000). A more integrated approach to control a specific plant-parasitic nematode may be necessary due to the high cost of fumigation and the ability of nematode populations to rebound after fumigation.

Crop rotation is a common cultural management practice used to reduce soilborne pests such as nematodes. Most nurseries rotate their production crops with cover crops (e.g., green manure crops) to increase soil organic matter, reduce compaction, and reduce pests. When damaging levels of a plant-parasitic nematode develops in a field, the nursery manager may have unknowingly used a host cover crop. For example, populations of *L. americanus* at a Georgia nursery continuously increased over a period of several years and damaged increasing numbers of loblolly pine seedlings. The field where the problem occurred had been used to test the feasibility of alternating production of loblolly pine seedlings with white oak (*Quercus alba* L.) seedlings (a host) instead of rotating to small grains (nonhosts), which were the normal cover crops used after seedling production (Cram and Fraedrich 2005). Similarly, *Tylenchorrhynchus ewingi* Hopper damaged pine seedling production at a Texas nursery where cowpeas (*Vigna unguiculata* L.), sorghum-sudan grass (*Sorghum bicolor* [L.] Moench), and rye (*Secale cerale* L.) were used as cover crops, all of which have been shown to be excellent hosts of *T. ewingi* (Fraedrich and others 2012). *Tylenchorhynchus* spp. such as *T. ewingi* and *T. claytoni* Steiner, that are found in some nurseries in the South, generally have wide host ranges that include sorghum-sudan grass, rye, corn (*Zea mays* L.), ryegrass (*Lolium multiflorum* Lam.), oats (*Avena sativa* L.), buckwheat (*Fagopryum esculentum* Moench), and various legumes (Cram and Fraedrich 2009, Fraedrich and others 2012). The common use of these species and other small-grain hosts for cover crops has probably made these plant-parasitic nematodes more difficult to control in some nurseries. Currently, the best, nonhost grain identified for control of *T. ewingi* and *T. claytoni* are certain varieties of pearl millet (*Pennisetum americanum* [L.] Leeke) (Johnson and Burton 1973, Cram and Fraedrich 2009, Fraedrich and others 2012). Pearl millet hybrids have been tested and bred for resistance to nematodes for many decades in the agriculture industry and various pearl millet cultivars have been reported to be resistant to

Paratrichodorus minor (Colbran) Siddiqi, *Meloidogyne* spp. *Belonolaimus longicaudatus* Rau, and *Pratylenchus brachyurus* (Godfrey) Filip. & Stek. (Johnson and Burton 1973, Timper and others 2002, Timper and Hanna 2005).

The practice of fallowing fields is an effective cultural practice to control plant-parasitic nematodes through starvation (Duncan and Noling 1998, Zasada and others 2010). Several field studies in forest nurseries have shown that fallowed fields kept weed free had significantly reduced nematode population densities. In the South, *L. americanus* and *T. claytoni* were controlled in fallowed fields within 1 year (Fraedrich and others 2005, Cram and Fraedrich 2009). In the North, Sutherland and Sluggett (1975) reported that corky root disease caused by *X. bakeri* could be controlled with fallow for 1 year and frequent disking during the summer months. Many nurseries can only afford a 1-year rotation with an alternate crop or fallow because of limited land base. The length of time it takes for a nematode population to decline to nondamaging levels in a fallow field or a field with a nonhost crop may determine which rotation option is best suited for the nursery.

Other nematode control methods, such as soil solarization, biofumigation, and steam treatments, have not proven reliable or practical for operational use (Zasada and others 2010). The use of solar heat has been tested in some nurseries and provided nematode control in one nursery, and controlled some fungi and weeds in several cases (Hildebrand 1989). Soil solarization works best in a hot climate where the soil can remain tarped for 4 to 6 weeks during the summer and where the soil temperatures over time reach a lethal level (Wang and McSorley 2008). One potential drawback of using solarization in forest nurseries is the failure of this practice to control heat tolerant fungi such as *Macrophomina phaseolina* (Tassi) Goid. (Mihail and Alcorn 1984, McCain and others 1986). The unpredictable nature of solarization to provide broad spectrum pest control and the need to apply the treatment over an extended period during summer months means that solarization is unlikely to replace fumigation and crop rotations for nematode control in most nurseries. In some individual cases, however, solarization could be useful when used in combination with other control practices.

Future Outlook for Nematode Control

Since the 1960s, many nurseries have relied primarily on fumigation with methyl bromide and several other fumigants to control nematodes in forest tree nurseries. The number of rules and regulations regarding the use of fumigants has been increasing in recent years, and forest nurseries are now adjusting to new regulatory changes enacted by the U.S. Environmental Protection Agency that have altered how and where fumigation can be applied (Zasada and others 2010). One of the greatest changes will be the buffer zone requirements, which are likely to reduce the area within nurseries that can be fumigated. The costs associated with fumigation also have been steadily increasing in recent years. Managers will need to rely on integrated strategies for suppressing nematode populations as they face changes in fumigation regulations. Practices such as cover cropping and fallow can be readily used to control many plant-parasitic nematode species, but more biological and ecological information is needed about the specific nematode species that cause problems in forest nurseries.

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