Chapter-13: Management of disease complexes

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Abstract: This chapter explores the core principles of managing nematode and plant disease complexes, exemplifying practical applications of management techniques for disease control and providing perspectives for enhancing future management strategies. The initial step involves the identification of the nematode/plant disease complex and understanding the biology of the organisms implicated. Subsequently, a comprehensive management program is formulated by assessing multiple potential components such as prevention, biological control, cultural practices, utilization of commercially available products, and constructing an integrated approach.

13.1 Introduction

This chapter delves into the fundamental principles of nematode and plant disease management, showcasing practical examples of how management techniques have been employed to control disease complexes and offering insights for the future advancement of management strategies. The initial phase of effective management involves the identification of plant parasitic nematodes and an understanding of their potential interactions with disease-causing agents such as fungi, bacteria, and viruses. Recognition typically begins when growers observe symptoms or indicators of crop distress. If a grower suspects the presence of nematodes or a disease issue, referencing a list of common symptoms and signs associated with nematode and disease problems often helps in discerning whether these manifestations may be attributed to other factors like suboptimal irrigation or fertilization practices. Once the pest complex present is identified, the next step involves evaluating various components of a comprehensive management program including prevention, physical control, biological control, cultural practices, and use of commercially available products (Bergeson, 1972; Hirano, 1975; Pitcher, 1978; Powell, 1971).

13.2 Symptoms and Signs of Disease Complexes

Some indicators suggestive of nematode infestation include reduced yield, premature maturity, delayed maturity, stunting, chlorosis, mid-day wilting, leaf drop, small fruit, yellowing, curling and twisting of leaves and stems, localized patches of stunted growth, and failure to respond to treatments targeting other issues, and unthriftiness. Due to the disruptive impact of nematode feeding and movement on plant vascular tissue, plants may struggle to extract sufficient water during hot periods, resulting in wilting. As the water stress diminishes by evening, plants regain vigor and may appear healthy again the next morning (Thorne, 1961; Westerdahl, 2011).

Following are some common symptoms of plant diseases. Leaf spot (dead, discolored, or injured areas with distinct margins on leaves or fruit). Blight (rapid yellowing, browning, collapse, and death of plant parts or the entire plant). Chlorosis (yellowing of normally green leaves and stems). Necrosis (browning or blackening of plant areas indicating cell death). Wilting (drooping of leaves, shoots, or the plant due to water stress). Distortion (abnormal traits or twisting of leaves, stems, or shoots). Mosaic (uneven patterns of yellow, light green, or dark green on leaves). Canker (dead area on stems or branches distinguished by color). Rot (breakdown and decay of plant tissue, often in roots and fruit). Dieback (death of leaf tips, shoots, or stems). Witches' broom (abnormal proliferation of shoots in a bushy appearance). Galling (swelling or abnormal growth on plant tissues). Stunting (abnormally small plant parts or a whole plant failure to grow to full size) (Agrios, 2005).

Upon suspicion of a problem, it is crucial to assess the available local resources that can support the development and execution of a management program, as these resources can vary significantly based on the geographical context. The WWW (World Wide Web) has developed into an excellent source of information for pest management-related information. Various entities such as federal and state environmental protection agencies, universities, and agricultural product companies have established online platforms offering valuable insights into pest management practices. The resources available may differ from one country to another. For instance, in the United States, state and federal environmental protection agencies host databases on integrated pest management accessible to the public. Moreover, entities like the United States Department of Agriculture (USDA), county agricultural commissioners, university departments, and Cooperative Extension programs provide expert advisors and researchers to aid in pest management efforts and operate diagnostic laboratories for identifying pest issues.

Those familiar with crop damage patterns would observe that many symptoms and signs observed are not exclusive indicators of nematode presence or plant disease and may stem from other underlying factors such as inadequate nutrition, irrigation practices, or alternative pathogens. This underscores that no definitive symptoms or signs exist to solely diagnose a disease complex issue. However, witnessing these symptoms and signs in the field should prompt suspicion of a potential disease complex, subsequently warranting further diagnosis through sampling.

13.3 Sampling Disease Complexes

The initial phase of effective management involves the identification of problematic nematodes or diseases existing within a system. Furnishing soil or plant samples to a diagnostic laboratory stands as a critical first step in this process. Once the identified organisms are determined, a crucial next step is to revisit the foundational biological aspects of nematodes and diseases, which is vital in formulating a robust management plan. In subsequent sections we will delve into the pertinent biological fundamentals essential for an effective management strategy.

Different methods are recommended for collecting nematode and plant disease samples to be submitted to a diagnostic lab. Due to the characteristic spotty, patchy, or nonrandom distribution of nematodes, obtaining an accurate representation of the nematode population typically involves collecting multiple smaller subsamples randomly and amalgamating them into a composite sample as opposed to taking a single sample from a singular location. The standard protocol for soil sampling includes the following steps: Excavate within the root zone where moisture is prevalent. Place the soil along with small roots into a plastic bag. Soil samples from various locations can be combined. Aim to collect approximately 1 kg of soil and roots. Healthy areas can also be sampled for comparison as well and placed in a separate bag. Seal the bags securely and store them in a cool environment (avoid freezing). Properly label the bags with essential information (e.g., name, address, sample location, date, crop history, current crop, next planned crop). Notify the designated laboratory of the sample that is being sent to them for analysis (McSorley, 1987; Wallace, 1978).

Determining the appropriate size of the sampling area is a crucial consideration influenced by several factors. The processing time required for each sample that encompasses extraction, counting, and reporting can vary from half an hour to several hours, with costs escalating accordingly. Given the labor-intensive nature of sample processing, it is imperative to recognize the value of investing the time to procure a "good" sample.

An informal survey conducted among applied nematologists reveals that most are confident in providing recommendations to growers based on samples obtained from a 2-hectare area. Nevertheless, due to the associated processing costs, growers frequently lean towards acquiring fewer samples from larger areas, often advocating for one sample from each 8-hectare block. In scenarios where economic constraints necessitate sampling larger areas, augmenting the number of subsamples within each sample can help to mitigate the potential loss of accuracy stemming from sampling a broader region. Prior to initiating the sampling process, it is essential to visually assess the site and identify any discernible variations such as differences in soil texture, soil moisture levels, drainage patterns, presence of healthy and unhealthy plant patches, or distinctive cropping histories that may necessitate the collection of separate samples. Each delineated area within the field is referred to as a stratum. For effective nematode population mapping and tailored management strategies, each stratum should be represented by a distinct sample (McSorley, 1987).

For plant disease sampling, to ensure accurate and timely plant disease diagnosis, it is crucial to submit high-quality plant or soil samples following these guidelines: Whole plants should be submitted, unless the issue is clearly related to leaves, stems, or fruits. When collecting samples, dig up plants carefully to keep the roots intact. Leave the soil attached to the roots to maintain plant viability during transit. Enclose roots and soil in a plastic bag near the base of the plants. Seedlings should be enclosed in a separate bag. Keep samples cool and away from direct sunlight. Clearly label sample bags on the outside of the bag with a permanent marker. Differentiate between affected and unaffected plants by packaging and labeling them separately. For plants up to 1-m high, submit a minimum of three whole plants with soil or 20 seedlings. For plants over 1-m high, separate the tops and bottoms before submission. Submit at least 10 whole plants or 20 seedlings (Agrios, 2005; Grogan, 1981; Wallace, 1978).

13.4 Diagnostic Procedures

As with sampling, different analysis techniques are utilized for nematodes than for plant diseases (Brown and Kerry, 1987). For nematodes, the choice of extraction technique depends on the nematode species of interest and the sample material, such as soil or plant tissue. Lab testing accuracy is enhanced by knowing the history of crops grown in the area, allowing for appropriate selection of extraction methods that target specific nematode genera. Different extraction methods yield varying proportions of nematode genera present in the sample, underscoring the importance of understanding the chosen lab's extraction protocol for accurate result interpretation. For example, looking at Fig 13.1, consider a sample containing lesion, ring, and dagger nematodes that is extracted by four different techniques: elutriator followed by sugar centrifugation, Baermann funnel lined with tissue, Baermann funnel lined with cheesecloth, and roots from the sample placed on a funnel in a mist chamber (Fig 13.1a). Relative shape and size of a nematode genus (Fig 13.1b) combined with activity level influence effectiveness of an extraction method. A sieving procedure is typically utilized to begin the process of extracting nematodes from soil (Fig 13.1c). The soil is mixed with a large volume of water and allowed to settle briefly for heavy particles to separate. The mixture is then poured through a series of sieves with mesh sizes tailored to retain larger debris and target nematodes based on their sizes and characteristics. Nematodes and soil particles collected on the sieves are backwashed into containers for further processing. The resulting solution, if clear enough, can be directly observed under a microscope or if not, subjected to additional purification steps for improved sample clarity. Methods used to improve clarity include placing soil on a Baermann funnel (Fig 13.1d) or elutriation followed by sugar centrifugation or flotation (Fig 13.1e). A misting chamber is utilized to extract nematodes from roots (Fig. 13.1f). The shape, size and activity level of a nematode species affects the relative number of nematodes that will be extracted by each method. Ectoparasitic nematodes residing outside plant roots in the soil won't be detected from root samples placed in a mist chamber, but endoparasitic root lesion nematodes will be. Each of the other three methods recovered lesion, ring, and dagger nematodes but in varying proportions. The relatively larger pore size of the cheesecloth lined funnel recovered more of the relatively long, thin dagger nematodes than did the other two methods. Stout, sluggish ring nematodes were best recovered by flotation in a sugar solution rather than having to actively move through pores in the funnel linings. More than twice as many lesion nematode were recovered by the Baermann funnel than by the sugar centrifugation method.

Fig 13.1. Methods for extracting nematodes from soil and roots: (a) relative proportions of lesion, ring, and dagger nematodes extracted by four different methods; (b) relative shapes and sizes of lesion, ring, and dagger nematodes; (c) sieving; (d) Baermann funnel; (e) elutriator; (f) mist chamber (author's own data, and images).

As with nematodes, relying solely on symptoms for diagnosis of plant diseases can be misleading, especially when different pathogens induce similar symptoms. Accurate diagnosis often requires identification of the causal agent, which may be microscopic and necessitates specialized equipment and techniques. Several different techniques are employed for diagnosing plant diseases. Light microscopy and histochemical staining are used for visual examination of pathogens. Isolation on artificial media is used to culture and identify pathogens. Soil extraction, electron microscopy, host range studies, and indicator plants may be needed for further analysis. Serological tests and other specialized procedures are used as needed for accurate identification (Agrios, 2005; Grogan, 1981; Wallace, 1978).

13.5 Biology of Nematodes as Related to Management

Once a diagnosis is obtained, the next step is to become familiar with the biology of the organisms involved as a prerequisite to developing a management program. This is especially true for utilizing nonchemical management techniques. Understanding the phylogenetic relationships of plant-parasitic nematodes has become crucial for devising effective nematode management strategies. The limited fossil evidence for nematodes initially led to a phylogenetic classification system primarily based on the morphology of species, traditionally studied using first light and then scanning electron microscopes. Advancements in biochemical and molecular analysis suggests that the evolution of nematodes parasitic on plants occurred independently four times within the phylum Nematoda resulting in four groups with distinct biological and morphological characteristics. These groups are the Triplonchida (Clade 1), the Dorylaimida (Clade 2), the Aphelelenchoididae (Clade 10) and the Tylenchida (Clade 12). Morphological evidence had previously also recognized these as distinct groupings (Baldwin, Nadler and Adams, 2004).

Characteristic of all plant-parasitic nematodes is that they utilize a stylet during their interactions with host plants. This stylet is used to puncture the host plant cell wall for nutrient extraction and, in some cases, to deliver secretory molecules into host cells to establish a permanent feeding site. Life cycle stage consist of an egg, four juvenile stages, and an adult. Molting occurs between each of the juvenile stages and the adult stage (Ferris and Ferris, 1998; Thorne, 1961).

Three basic life history patterns are found among plant parasites: migratory ectoparasitic, migratory endoparasitic and sedentary endoparasitic. All life cycle stages of migratory ectoparasites are present in the soil, while eggs, juveniles, and adults of migratory endoparasites can be found either in roots or the soil. In sedentary endoparasites, the second-stage juvenile is typically the migratory and infective stage that emerges from the egg, resides in the soil, and penetrates the root to establish a feeding site. Subsequent juvenile stages and adults of sedentary endoparasites are generally nonmotile and remain fixed at a single feeding location. Eggs may be located within roots or in the soil. Categorizing nematodes based on their life history habits can offer valuable insights when exploring management strategies. For instance, when considering the application of chemicals or soil amendments, such products are more likely to target nematodes in the soil (migratory ectoparasites) rather than those residing within roots, unless the product has systemic effects. Despite spending some time in the soil during their life cycle, a substantial portion of endoparasites is typically found within roots (Ferris and Ferris, 1998; Thorne, 1961; Westerdahl, 2011).

Nematodes damage roots in several ways. Mechanical injury occurs by penetration and movement through plant tissues leading to cell death or stimulating cell growth. Cellular changes occur by induction of necrosis (death of cells) and alterations in cell growth. Feeding in vascular tissues disrupts the flow of water, nutrients, and food, impacting the plant's overall health. Infection is facilitated by creation of entry points for other microorganisms like bacteria and fungi. Nematodes can introduce pathogens either on their bodies or within their bodies, or they can aid the entry of pathogens that are already present on the plant cell surface. Transmission of pathogens including viruses. Stress from nematode feeding leads to heightened vulnerability of plants to environmental stress factors. Nematodes are adept at surviving as obligate parasites, as they have evolved over millions of years to stress their plant hosts rather than kill them outright (Thorne, 1961).

Nematodes play a crucial role in the transmission of viruses in various plant species. Nematodes responsible for transmitting plant viruses are found in the Triplonchida and the

Dorylaimida. Triplonchida facilitate the transmission of tubular viruses known as tobraviruses. Dorylaimida vector polyhedral-shaped or isometric viruses referred to as nepoviruses. The process of virus acquisition by nematodes occurs during their feeding on infected plants. Some viruses can be acquired in as little as a 5 to 15-minute feeding session, while other viruses may necessitate up to a 24-hour feeding period. Nematode vectors can acquire the virus instantly upon ingesting cell contents from infected roots, with no latent period within the nematode before transmission. Viruses transmitted by nematodes do not persist through nematode molting stages and do not transfer through nematode eggs. Both adult and juvenile stages of nematode vectors have the capacity to transmit associated plant viruses. Virus transmission between nematode vectors and specific virus strains is remarkably specific and appears to be guided by interactions between the virus protein coat and the nematode feeding apparatus lining (Alfaro and Goheen, 1974; Bergeson, et al., 1964; Christie and Perry, 1951; Das and Raski, 1968; Esmenjaud et al., 1993; Harrison et al., 1971; Harrison, 1978; Hewitt, Raski, and Goheen, 1958; Lamberti and Roca, 1987; McGuire, 1973; Taylor and Brown, 1981).

Plant parasitic nematodes play a crucial role in the development of diseases in plants. They can act independently as pathogens or engage in synergistic relationships with other plant pathogens, thereby leading to the formation of complex disease conditions. Interactions between nematodes and various wilt-inducing and root-rot fungi are commonplace in agricultural settings, underscoring their significance in influencing disease dynamics. Within Tylenchida, the following fungal disease complexes are recognized: *Meloidogyne (Alternaria, Fusarium, Rhizoctonia, Phytophthora, Phomopsis, Sclerotium), Pratylenchus (Pythium,Rhizoctonia, Verticillium),* Trichodoridae *(Rhizoctonia),* Heteroderidae *(Rhizoctonia), Rotylenchulus (Fusarium, Rhizoctonia)*, and *Belonolaimus (Fusarium).* Also, in Tylenchida, the following bacteria diseases complexes are found: *Anguina (Clavibacter), Ditylenchus (Pseudomonas, Clavibacter), Helicotylenchus (Pseudomonas), Meloidogyne (Pseudomonas, Curtobacter, Agrobacterium, Clavibacter), Pratylenchus (Pseudomonas, Agribacterium), Rotylenchulus (Agrobacterium), Meloidogyne (Ralstonia),* and *Criconemoides (Pseudomonas).* Within the Aphelelenchoididae, are found the bacteria disease complexes: *Aphelenchoides (Rhodococcus, Clavibacter*) (Agrios, 2005; Sankaralingam and McGawley, 1994; Sikora and Carter, 1987; Starr et al., 1996).

Plant parasitic nematodes can serve as vectors for other pathogens and also inflict wounds on plants, compromising their defense mechanisms and rendering them highly susceptible to disease development. By augmenting the host susceptibility, nematodes contribute significantly to the acceleration and severity of wilt and fungal rot diseases in affected plants. Evidence suggests that root exudates from plants infected with root knot nematodes not only stimulate the growth of fungal pathogens but also suppress the population of actinomycetes, known antagonists of wilt-causing fungi like *Fusarium* spp. The physiological alterations induced by root knot nematode infection further facilitate fungal penetration into the plant tissues, fostering wilt progression. Plant parasitic nematodes exert a profound impact on the physical and chemical defense mechanisms of plants through intricate interactions at the cellular and biochemical levels. Upon infestation, nematodes establish feeding sites within the xylem parenchyma cells, eliciting substantial alterations in the plant's morphology, anatomy, and biochemistry. Root knot nematodes induce the formation of giant cells in plants, which exhibit sustained metabolic activity due to continuous stimulation by the nematode. These giant cells exhibit elevated concentrations of sugars, hemi-cellulose, organic acids, free amino acids, proteins, and lipids, creating a nutritive environment conducive to the proliferation of fungal pathogens. The persistence of these giant cells in an immature state delays the maturation and suberization of other vascular tissues, facilitating the unrestricted penetration and establishment of fungi like *Fusarium* within the xylem elements. Consequently, the nematode-mediated inhibition of phytoalexins production compromises the plant's resistance to wilt-causing fungi, further exacerbating disease susceptibility (Khan and Pathak, 1993; Webster, J.M., 1985).

13.6 Biology of Plant Diseases as Related to Management

The definition of plant disease can vary, but it is commonly characterized as any condition in a plant that disrupts its normal growth and development, caused by living and nonliving agents. Plant disease is an anomaly that hampers the natural growth and development of a plant, arising from both living and nonliving agents. The concept of the disease triangle is a fundamental principle in plant pathology that illustrates the conditions necessary for disease occurrence. Each of three equal sides of the triangle represents a factor crucial for disease development. One side of the triangle signifies the host plant. It refers to the plant species or variety that is susceptible to the disease. The second side represents the factor or organism responsible for initiating the disease. The third side depicts the environmental conditions necessary for the interaction between the host plant and the causal agent to lead to disease development. This can include factors like temperature, humidity, or other elements conducive to disease progression. Disease occurs when all three components of the triangle are present and interact. If any of the three factors is missing or inadequate, the triangle collapses, preventing disease development. For instance, unfavorable environmental conditions may inhibit disease progression even if the host plant and causal agent are present. Understanding the disease triangle concept aids in disease management strategies by targeting one or more sides of the triangle to disrupt disease progression (Agrios, 2005).

Fungi and oomycetes represent the largest group among plant pathogens, responsible for the majority of plant diseases. Despite the vast number of fungal species only a small fraction are pathogenic to plants, causing notable agricultural and horticultural losses worldwide. These usually microscopic organisms consist of hyphae, threadlike structures that form webs or mycelium within plant tissues. Fungi primarily reproduce and spread through spores, with different types of spores influencing disease dissemination and management strategies. Fungi infect plants through direct penetration of tissues or via natural openings like stomata, hydathodes, and wounds. The presence of free surface water on plants is often essential for fungal infection, making fungal diseases more prevalent after wet periods or when overhead irrigation is practiced. Fungi are commonly dispersed through wind, splashing water (from rain or irrigation), insects, and human activities. Cultural practices contribute to fungal spread, such as through contaminated tools like pruning shears, pots, or soil. Understanding the biology, reproduction methods, infection pathways, and dispersion mechanisms of fungal pathogens is crucial for implementing effective disease management strategies. Recognition of the environmental conditions favoring fungal infections, as well as the various modes of transmission, can guide preventive measures to mitigate disease outbreaks. Adoption of integrated pest management practices that consider both cultural and chemical control methods can help limit fungal disease impact and promote plant health in agricultural and horticultural settings (Evans and Haydock, 1993).

While fungal cell walls are predominantly composed of chitin, cell walls of oomycetes or water molds which includes *Phytophthora and Pythium* consist mainly of cellulose. *Phytophthora* have the capacity to impose significant economic losses on crops globally. Control of plant diseases caused by *Phytophthora* is often challenging with chemical means, leading to the primary management strategy of developing resistant cultivars. *Pythium* induced root rot is a prevalent crop disease, with damping off occurring when the organism targets newly emerged or emerging seedlings. *Pythium* wilt, another manifestation of the disease, occurs when older plants are infected by zoospores. *Pythium* species are known to be generalists with a broad range of hosts, while *Phytophthora* species tend to be more host-specific. The survival of *Pythium* species is prolonged by their capacity to function effectively as saprotrophs, allowing them to persist for extended periods on decaying plant material. This attribute makes eradicating the pathogen through crop rotation alone challenging (Farrar, Nunez and Davis, 2002; Vivoda et al., 1991; White, 1996).

Verticillium is a genus of fungi within the division Ascomycota. At least five species within *Verticillium* are known to induce a wilt disease in plants referred to as *Verticillium* wilt. The disease poses challenges in management due to various characteristics inherent to *Verticillium* fungi, such as its ability to persist in soil for extended periods without a host, a broad host range, and the limited resistance found in host germplasm. The fungus survives predominantly in the soil in the form of microsclerotia, entering the plant via the root system, where it then colonizes the plant's vascular system, often leading to the eventual death of the plant (Conroy, Green, and Ferris, 1972; Francl and Wheeler, 1993; Hasan and Khan, 1985; MacGuidwin and Rouse, 1990; Powelson and Rowe, 1993).

Fusarium is a genus of filamentous fungi categorized as hyphomycetes, commonly found in soil and closely associated with plants. This genus encompasses several economically significant plant pathogenic species. While primarily saprotrophic, certain *Rhizoctonia* species act as facultative plant pathogens, leading to significant crop diseases of commercial importance

(El-Sherif, 1991; France and Abawi, 1994; McLean and Lawrence, 1995; VanGundy, Kirkpatrick, and Golden, 1977).

Bacteria are single-celled microorganisms lacking an organized nucleus. Only a small proportion of naturally occurring bacteria possess the ability to invade plants and induce disease. Bacteria primarily reproduce through cell division, which can lead to rapid population growth within plant tissues. Bacterial presence and propagation within plants often occur without visible signs initially, making early detection challenging. Bacteria typically cannot penetrate intact and healthy plant tissues. Infections commonly result from wounds or openings in plants. Bacterial pathogens spread through various means, including splashing water (from rain or irrigation), insect vectors, and cultural practices. Contaminated tools, plant debris, or soil in pots can serve as sources of bacterial dissemination in agricultural and horticultural settings. Seed transmission is another route through which bacteria can be transmitted from infected plants to progeny (El-Sherif, 1991; Khan and Pathak, 1993; Sikora and Carter, 1987, Sitaramaiah and Pathak, 1993).

The link between ring nematode and bacterial canker poses a significant threat to various fruit tree species such as almond, apricot, cherry, kiwi, nectarine, peach, pear, plum, and prune. Orchards typically harbor the bacteria *Pseudomonas syringae*, while the presence of the ring nematode (*Criconemoides* sp.) can stress trees, making them more susceptible to bacterial canker infection. This stress is especially pronounced in younger trees grown in sandy soils.
Stressed stone fruit and nut trees due to ring nematode infestation are more prone to bacterial canker by *Pseudomonas syringae* than healthy trees. Symptoms of bacterial canker, manifesting mostly in spring, include dead branches and lesions with dark-colored sap exudation. The disease can spread uniformly across an orchard or concentrate in localized areas known as "bacterial canker holes." Its severity varies from minor twig damage to the death of whole trees, with

instances of replanted trees succumbing to recurring infections. A notable connection was observed between low fertilization levels and bacterial canker development in an orchard unaffected by ring nematode infestation (Mai and Abawi, 1981).

Phytoplasmas are specialized bacteria that lack cell walls, possessing unique characteristics compared to typical bacteria. These microorganisms are obligate parasites that are associated with diseases in plants and can be transmitted through insect vectors, impacting plant health and productivity. They are not known to be transmitted by nematodes (Agrios, 2005).

Plant health issues caused by nonliving factors are commonly referred to as disorders when discussing abiotic agents. The distinction between disorders and diseases lies in terminology, where disorders typically indicate nonliving causal factors, while diseases suggest living agents are responsible. Abiotic agents affecting plant health can be broadly classified into cultural factors related to human practices and environmental factors associated with natural conditions. Cultural factors include human activities such as improper planting practices, over or under watering, nutrient deficiencies or excesses, soil compaction, pesticide misuse, and other management practices that can impact plant health. Environmental factors include natural elements like temperature fluctuations, light exposure, humidity levels, soil composition, air pollution, and other nonliving aspects of the plant's surroundings that can influence its growth and development (Agrios, 2005).

13.7 Prevention

Some key techniques to minimize the spread of pests include implementing quarantine measures that can help restrict the movement of potentially infested plants, plant materials, or soil to prevent the introduction and spread of pests in new areas; using certified planting materials that are inspected and tested to ensure they are free from known pests and diseases;

checking suspect materials before planting; clearing debris of annual plants and trimming perennials after frost to reduce overwintering sources of diseases; removing spent flowers and leaf debris throughout the growing season to reduce disease inoculum buildup and limit disease spread; properly composting manure before application to help kill weed seeds, pathogens, and pest eggs; cleaning equipment after use including disinfesting pruning tools, pots, and flats; and avoiding contaminated irrigation water (Maas, 1987).

The limited awareness of nematodes and pathogens among the public can lead to the inadvertent introduction of pests into new environments, particularly through the importation of ornamental plants. Growers who have not encountered significant pest issues may lack awareness of the potential threats posed by pests like nematodes and pathogens. This lack of awareness can result in the unintentional movement of pests through various means such as soil, irrigation water, equipment, and planting stock. In agricultural settings such as farms, nurseries, and government agencies, there is often a frequent turnover of personnel. This personnel turnover can contribute to a lack of consistent knowledge and understanding of pest management practices. Therefore, there is a continuous need for education and training on the importance of prevention in managing pest complexes effectively.

Government agencies recognize the significance of both external programs aimed at managing plants imported into a region and internal programs focused on safeguarding nursery stock cultivated within the region, which may be intended for commercial farm planting or export. During periods of financial constraints, government personnel may mistakenly assume that a program detecting only a few positive findings each year is no longer necessary. However, effective internal and external sanitation programs play a critical role in preventing the entry of destructive pests and pathogens into protected areas and confining them to limited hectares. A practical illustration of this concept can be seen by considering a nursery with common row and plant spacing practices. For instance, if a nursery employs a typical row spacing of 1-meter with plants placed 15-cm apart, and approximately half of these plants reach harvestable size, an area of 0.5-hectares containing infested nursery stock could result in the infestation of over 40-hectares of farmland if the farmland is planted with 300 trees per hectare (McNamara, 1995).

Washing soil off equipment before moving it to a new, uninfested field can help prevent the inadvertent spread of nematodes and other pests, reducing the likelihood of infestations in previously unaffected areas. The potential for nematodes to be transmitted through irrigation water poses a significant risk to agricultural systems. Research conducted in Washington state exemplifies how irrigation canals can serve as vectors for the transportation of various genera of plant parasitic nematodes. These studies not only confirmed the presence of multiple nematode species in irrigation water but also demonstrated their viability through greenhouse experiments. It was estimated that during a typical irrigation cycle, growers could unknowingly introduce several million parasitic nematodes into their fields through irrigation water. One practical solution involves the use of settling ponds in conjunction with irrigation practices. By diverting irrigation water into settling ponds before application, growers can allow nematodes and other sediment to settle out, a process that typically only requires a few minutes. Subsequently, water can be drawn off from the top of the settling ponds for irrigation purposes. There is also potential for nematodes and pathogens to be spread through various less-studied avenues, such as wind, birds, and other animals (Esser, 1984; Faulkner and Bolander, 1966).

13.8 Physical Methods

Physical methods play a crucial role in nematode management, often complementing cultural and chemical control strategies. These methods offer effective and environmentally friendly approaches to combat nematode infestations. Some key physical methods utilized in management of nematodes and plant pathogens include heat treatment of planting stock, steam sterilization of soil, soil solarization, and root pruning of infected plant material before planting (Maas, 1987).

13.8.1 Heat Treatments

Heat treatment of planting stock involves subjecting planting materials to elevated temperatures to eliminate nematodes and other pathogens. This method has demonstrated effectiveness in controlling nematodes and pathogens in various crops and has been particularly valuable in commercial settings, targeting specific species infesting different plant types. Hot water treatments of planting stock have been a longstanding management practice, dating back to at least the early 1900s. Chemicals may be added to a hot water treatment bath to enhance effectiveness against nematodes, fungi and bacteria. Examples include the use of hot water treatments for managing stem and bulb nematodes in daffodils and garlic, root-knot nematodes in grape rootstocks, and foliar nematodes in Easter lilies (Bridge, 1975; Salch, Abu-Gharbieh, and Al-Banna, 1988).

For hot water treatments to be successful, precise time and temperature controls are essential. It is critical to determine the optimal combination of temperature and exposure duration that effectively eradicates target organisms while minimizing damage to the plant material. The susceptibility of each pest species and crop variety must be assessed on a small scale prior to implementing large-scale treatment programs to ensure efficacy and prevent potential harm to plants. Using inappropriate temperatures or exposing the plant material for either too long or too short a period can compromise the treatment's effectiveness. Therefore, it is imperative to conduct preliminary testing to establish suitable time-temperature parameters tailored to specific pest-crop combinations. Existing literature provides valuable information on recommended temperature and duration ranges for hot water treatments against different nematodes in various crops, serving as a useful reference for experimental testing and treatment optimization (Bridge, 1975; Noling, 1994).

Examples of temperature and duration guidelines for hot water treatments include 51.7°C for 5 minutes for root-knot nematodes on grapevines, 48.9°C for 20 minutes for stem and bulb nematodes on garlic, and 43.9-44.4°C for 180 minutes for stem and bulb nematodes on daffodils. These specific parameters serve as starting points for customization and refinement based on the target nematode species and crop requirements. Some procedures incorporate pretreatment tanks to partially raise the planting stock to the required temperature, especially if the stock is being transferred from cold storage or soil before treatment. Cooling stock promptly after treatment is crucial. Additional tanks for cooling or hosing down treated stock with cool water post-treatment may be necessary to prevent damage to plants. Maintaining precise control over the temperature and volume dynamics is crucial when conducting hot water treatments to effectively manage nematodes and pathogens. Ensuring a large enough volume of water compared to the planting stock helps prevent significant temperature drops when stock is added. Shorter treatment times require quicker temperature stabilization to maintain the desired treatment levels. For treatments lasting, for example, 5 minutes, the temperature should ideally return to the treatment level within 1 minute (Bridge, 1975).

13.8.2 Steam

Utilizing steam to heat soil to temperatures ranging between 66°C to 71°C for a dwell time of 20 minutes has proven effective in eradicating weed seeds and nutsedge tubers in the soil, along with combating nematodes and other pathogens. Recent evaluations of steam applicators in crops including carrot, lettuce, spinach, and strawberry have shown promising results against pathogens such as *Sclerotinia minor, Phytophthora* spp., *Pythium ultimum*, *Verticillium dahliae*, and suppression of *Fusarium* spp. Steam applicators are designed to move continuously to prevent damage to beneficial soil microorganisms. Rapidly heating soil to 66 to 71°C within 60-90 seconds typically sustains the required high temperature for the 20-minute dwell time above 70°C. Prior to planting, steam is applied in narrow bands at depths of 8-13 cm (Kim, Kim, and Fennimore, 2021).

Soils and planting mixes for potting can be effectively steam-treated in enclosed containers, such as steam chambers, to eliminate pests and pathogens. Treatment of raised beds can be accomplished through methods like buried perforated tubing or pipes. Another approach involves blowing steam under a plastic sheet secured at the edges to treat the soil. Perforated tubing is typically buried 25 to 80 cm deep, influencing the area that can be effectively treated from a single tube. Steam treatments for larger areas usually require 4 to 8 hours to complete, aiming to achieve temperatures of 60°C to 70°C for a minimum of 30 minutes for effective control (Fennimore et al., 2014; Guerra et al., 2022; Kim, Kim, and Fennimore, 2021).

13.8.3 Soil Solarization

Soil solarization involves heating the soil through solar energy by covering it with clear plastic sheets. This method raises soil temperatures to levels that can effectively kill nematodes and pathogens present in the top layers of the soil. The process involves using plastic mulches to generate lethal temperatures in the soil and is currently being employed in various countries to combat root knot and soil-borne diseases (Chellemi et al., 1993; Gamliel and Stapleton, 1993; Katan 1981; Noling, 1994; Stapleton and DeVay, 1986; Stapleton, Lear, and DeVay, 1987).

Soil solarization typically requires 4 to 6 weeks of coverage, preferably during the hottest time of the year when maximum solar energy can be harnessed for heating the soil. Soil solarization has demonstrated effectiveness in controlling weeds and fungal pathogens due to the elevated temperatures reached during the process, which can significantly reduce their populations in the soil. Soil solarization may not be as effective for nematodes compared to weeds and fungi due to the limited depth of treatment effectiveness. Nematodes residing deeper in the soil profile may not be as impacted by the solarization process. Soil solarization has been proven effective in controlling root knot nematode, *Verticillium* wilt, and weeds in crops (Overman and Jones, 1986; Stapleton and DeVay, 1986; Salch, Abu-Gharbieh, and Al-Banna, 1988).

13.8.4 Remote Sensing

Advances in remote sensing technologies, such as infrared and digital thermography, have enabled the detection of areas within fields where plant parasitic nematodes are causing damage. By integrating this technology with precision farming equipment, it is possible to improve control efficacy by precisely applying nematicides in high nematode density areas (Nutter et al., 2002).

13.8.5 Root Pruning

Root pruning is a technique that involves removing nematode-infested roots from plant material, such as bulbs, before planting. Removing infested roots prior to planting, especially in bulb crops where nematodes like lesion nematodes may predominantly infest the roots, has shown some promise in reducing nematode populations in roots and subsequently improving bulb growth. This practice helps prevent the introduction of nematodes into the soil and reduces the risk of nematode spread to healthy plants during transplantation (Westerdahl et al., 1998).

13.9 Biological Control

13.9.1 Basic Concepts

Classical biological controls employ living organisms that function as competitors or antagonists to the disease-causing agent. Biological control in the context of nematode and pathogen management encompasses a range of methods, including the use of predators, parasites, soil amendments, toxins produced by microorganisms, killed microbial agents, and natural products. Resource competition, interference, and occupation of space contribute to success of biological control. Living organisms compete for resources, preventing the disease agent from accessing essential nutrients. Some organisms inhibit the growth and development of pathogens through the production of inhibitory compounds. Only one organism can occupy a given space, restricting the growth of pathogens in that area. A number of "natural" products and soil amendments are available that claim to impact nematode populations indirectly, through testimonials or implicit suggestions rather than explicit claims. These products are not classified as nematicides but are marketed based on their ability to enhance soil conditions for plant growth while potentially affecting nematodes as well (Sikora, Bridge, and Starr, 2005; Stirling, 1991).

The observation of population cycling dynamics in parasite-host relationships, where parasites selectively target a portion of the host population, has been a subject of study in ecological research. Over evolutionary time scales, parasites and hosts engage in co-evolutionary interactions that shape their relationship. Well-adapted parasites develop strategies to exploit specific host populations while minimizing the risk of host extinction, maintaining a stable and sustainable coexistence. Host-specific parasites face the risk of host extinction if they excessively parasitize their hosts. This can limit the effectiveness of using a single parasite for biological control. The challenges associated with using host-specific organisms have led to skepticism about the practicality of natural biological control in certain contexts. While relying solely on a single organism may not be feasible, the potential for developing combinations of organisms or integrating biological control agents with cultural practices offers promising avenues for enhancing management outcomes. Various soil-dwelling predators, including mites, tardigrades, turbellarians, enchytraeids, insects, and predatory nematodes, have demonstrated potential in providing some level of control over plant parasitic nematodes in both natural and agricultural environments (Kerry, 1990; Tedford et al., 1993)

13.9.2 Fungal Parasites

A wide range of nematophagous fungi have been identified. The categorization of nematophagous fungi based on their feeding habits rather than phylogeny provides insights into the diverse mechanisms employed by these fungi to trap and feed on nematodes. Nematode trapping fungi are classified into various groups based on their feeding habits, such as adhesive networks or knobs, nonconstricting or constricting rings, or adhesive conidia. Each group employs distinct structures and mechanisms to adhere to and attack nematodes, reflecting the diversity of strategies evolved by these fungi to capture and feed on nematodes. For nematode trapping fungi, the trapping process is often considered a passive activity, where the fungus awaits the presence of nematodes to come into contact with the adhesive networks, knobs, rings, or conidia. Once a nematode interacts with these structures, they may become stuck or wedged, leading to subsequent fungal penetration and utilization of the nematode as a nutrient source.: Studies have shown that nematodes tend to put their heads through constricting rings, potentially leading to entrapment or adherence to adhesive substances. The formation of more rings by fungi in the presence of nematodes suggests a responsive mechanism to increase trapping efficiency. The exact reasons why nematodes engage with these structures, such as putting their heads through rings, are not fully understood but likely involve a combination of innate behavior and environmental cues. Once nematodes are trapped, fungal hyphae penetrate the nematode's body, leading to nutrient extraction and utilization by the fungus (Kerry, Crump, and Mullen, 1982).

Some nematophagous fungi have been observed to be poor competitors in the natural environment. They rely on the nematode cuticle for protection from other organisms. This indicates that these fungi have evolved specialized adaptations to exploit nematodes as a food source while potentially facing challenges in competing with other microorganisms in their ecological niche. To enhance the field application of nematode parasitic fungi for biological control purposes, some fungi have been encapsulated in pellets made from materials like calcium alginate. These pellets serve multiple purposes: they aid in the dispersal of the fungi in the field, protect the fungi from environmental stresses, and provide a sustained food source for the fungi until they encounter nematodes (Becker and Schwinn, 1993).

Zoosporic fungi represent a diverse group of fungi characterized by their motile spores, known as zoospores, which have the ability to actively swim through aqueous environments. These fungi have evolved unique strategies to target nematodes, harnessing their swimming capability to seek out potential hosts, attach to their cuticles, and subsequently develop hyphae that feed on the nematodes. Zoosporic fungi exhibit a broad spectrum of host specificity, parasitizing various life stages of nematodes. Some fungi target vermiform adult or larval nematodes, while others specialize in parasitizing nematode eggs or cysts. This diversity in host specificity allows zoosporic fungi to exploit different nematode populations and life stages as potential sources of nutrients and reproduction. Within the group of nematode egg parasites, zoosporic fungi encompass a wide range of taxonomic groups, reflecting the evolutionary diversity and adaptability of these fungi in targeting nematode eggs for parasitic interactions. By parasitizing nematode eggs, these fungi disrupt the reproductive cycle of nematodes, potentially reducing nematode populations and impacting their overall abundance in the environment (Becker and Schwinn, 1993; Stirling, 1991).

13.9.3 Bacteria

Three bacteria *Pasteuria penetrans*, *Bacillus thuringiensis*, and *Streptomyces avermitilus*, have all been extensively studied in relation to nematodes. *P. penetrans* is classified as a true parasite of nematodes. It infects nematodes by attaching to and penetrating their cuticles, subsequently colonizing the nematode's body and ultimately leading to the nematode's death. *B. thuringiensis* is known for producing insecticidal proteins known as crystal toxins (Cry toxins) that target specific insect pests by causing gut paralysis and ultimately leading to their death (Becker and Schwinn, 1993; Kerry, 1995: Stirling, 1991).

While *B. thuringiensis* primarily targets insects, some nematode species have been found to be susceptible to certain Cry toxins produced by this bacterium. This has led to interest in exploring the potential of *B. thuringiensis* as a biocontrol agent for nematodes. *S. avermitilus* is known for producing avermectins, a class of compounds with anthelmintic properties that target parasitic worms, including nematodes. Avermectins produced by *S. avermitilus* have been widely used in veterinary and agricultural applications to control parasitic nematodes in livestock and crops. These compounds disrupt nematode nervous systems, leading to paralysis and death (Kerry, 1995: Stirling, 1991).

While *Pasteuria penetrans* acts as a direct parasite of nematodes, *Bacillus thuringiensis* and *Streptomyces avermitilus* utilize toxins or metabolites to affect nematodes indirectly by causing toxicity or physiological disruptions. *Pasteuria penetrans*, an actinomycete bacterium was initially misidentified as a protozoan, *P. penetrans* was first described by Thorne in 1940 and has since been the subject of numerous studies involving the isolation and characterization of different strains. Spores of *P. penetrans* adhere to the cuticle of a nematode, establishing initial contact for infection. Subsequently, a penetration tube forms, facilitating the entry of the bacterium into the nematode host. Once inside the nematode, *P. penetrans* undergoes reproduction, utilizing the nematode's cuticle as a protective environment. This intracellular replication leads to the formation of a considerable number of spores within the nematode host. While the nematode may initially survive and continue to feed post-infection, the reproductive capacity of the nematode is significantly impaired. The presence and proliferation of *P. penetrans* within the nematode host disrupt its normal physiology and reproduction, leading to a decrease in nematode population dynamics. By reducing the reproductive output of infected nematodes and potentially causing mortality over time, *P. penetrans* contributes to the regulation of nematode numbers in various ecosystems. Studies have shown that a single nematode infected with *P. penetrans* can harbor a vast number of spores, with estimates reaching up to two million spores within an individual nematode (Kerry, 1995: Stirling, 1991).

Avermectin (MK-936) is a potent macrocyclic lactone compound derived from the actinomycete bacterium *Streptomyces avermitilus* through fermentation processes. In insects, avermectin exerts its insecticidal effects by blocking the neurotransmitter gamma-aminobutyric acid (GABA). This disruption of GABA function leads to paralysis and death in susceptible insects. The acute oral toxicity of a 0.15 EC formulation of avermectin in rats is reported to be 650 mg/kg, indicating a moderate level of toxicity. Additionally, acute dermal toxicity studies in rabbits showed a relatively low toxicity level with a reported value of 2000 mg/kg. Avermectin is widely utilized in various commercial formulations for insect control. Its effectiveness at very low dosages makes it a preferred choice for integrated pest management strategies, providing efficient control against a wide range of insect pests. Avermectin has also gained popularity for its efficacy in controlling nematode parasites in animals. It has shown effectiveness against various animal parasitic nematodes, contributing significantly to the management of parasitic infections in livestock and pets. While avermectin has demonstrated activity against plant parasitic nematodes, its practical use in plant nematode control is limited due to the molecule's relatively large size, which hinders its movement through the soil (Kerry, 1995: Stirling, 1991).

13.9.4 Suppressive Soils

The concept of suppressive soils, defined as soils that should theoretically have nematode issues but do not, has captivated nematologists for an extended period. Research has indicated that soils exhibiting suppression against sugar beet cyst nematodes often harbor one or more species of nematode-parasitic fungi. It was discovered that *H. rhossiliensis*, a species parasitic on juvenile stages of sugar beet cyst nematode, was prevalent in sugar beet fields across California, including a high occurrence rate in San Joaquin County where it was found in 80% of sampled fields. Additionally, other nematode-parasitic fungi such as the ring trapping fungus *Arthrobotrys dactyloides*, as well as species parasitic on cysts and eggs of sugar beet cyst nematode like *Hyalorbilia oviparasitica*, *Dactylella oviparasitica*, and *Brachypyoris oviparasitica*, are commonly present in fields with a history of sugar beet cultivation. It has been shown that nematode parasitic fungi potentially contribute to the observed declines in nematode populations within suppressive soils. It has been proposed that the populations of parasitic fungi act in a self-regulating manner, maintaining a level below that which would completely eliminate the nematode population. This self-regulation ensures that a sustainable food source for the fungi, in the form of nematodes, remains available. The interplay between nematode-parasitic fungi and nematode populations in these suppressive soils suggests a complex and finely balanced ecological dynamic that contributes to the mitigation of nematode-related issues in agricultural systems. At harvest in chemical control field trials, it is not uncommon to observe higher nematode populations in chemically treated plots compared to untreated controls. This difference can be misinterpreted as the untreated controls having naturally suppressive soils. However, the increased nematode populations in chemically treated plots may be attributed to healthier root systems that can support larger nematode populations compared to those in untreated plots. The difference in nematode populations between chemically treated and untreated plots in field trials may not necessarily indicate natural suppression in the untreated controls (Chen et al., 2021; Gair, Mathias, and Harvey, 1969; Jaffee et al., 1992; Kerry, 1995; Tedford et al., 1993).

13.9.5 Soil Amendments

Soil amendments can act through different mechanisms to help suppress nematodes and enhance overall soil health. Addition of beneficial microorganisms can compete with nematodes for resources or produce compounds that are harmful to nematodes. Some soil amendments can promote the growth of nematophagous fungi that actively feed on nematodes. Certain products are designed to reintroduce beneficial microorganisms back into the soil after their depletion by chemical treatments. Amendments that enhance soil structure and water-holding capacity can create conditions less favorable for nematodes. Nutrients that reduce plant stress in nematode-infested soils can help plants better withstand nematode damage. Adding organic matter to soils offers several advantages, including improved soil structure, enhanced water retention, and nutrient provision. These enhancements reduce plant stress, potentially mitigating the impact of stress induced by plant parasitic nematodes. Some amendments can release compounds that have nematicidal properties, either directly or after breakdown in the soil. Soil amendments have been sourced from various materials, including waste products like coffee grounds, newsprint, crab shells, and quinoa bran. Substances containing chitin, like crab and shrimp shells, are believed to stimulate the growth of nematophagous fungi. These fungi utilize chitin present in nematode eggshells as a food source. Additionally, specific crops such as marigolds, vetch, and sesame can be grown for their potential nematode-suppressive properties. Decomposing plant-based products like marigolds, brassicas, and sesame release chemicals with nematicidal properties. For instance, brassicas have been found to release compounds similar to those in chemical nematicides like metam-sodium-containing products (Kaplan and Noe, 1993; Rodriguez-Kabana, 1986; Spiegel, Chet, and Cohn, 1987; Culbreath, Rodriguez-Kabana, and Morgan-Jones, 1985; Stapleton, Duncan, and Johnson, 1998; Westerdahl et al., 1992).

The mode of action of many natural soil amendments remains unclear. Due to the variability in agricultural soils, the effectiveness of these amendments may vary across different farming scenarios. In developing treatment programs with soil amendments, it is crucial to include untreated replicated areas for comparison. By leaving some sections untreated, growers can accurately assess the success of the treatment program and adjust strategies as needed based on observed outcomes (Muller and Gooch, 1982; Rodriguez-Kabana, Morgan-Jones, and Chet, 1987).

13.9.6 Commercially Available Products

Commercially available biological nematicide active ingredients include: *Myrothecium verrucariae* strain AARC-0255 (a toxin produced by a fungus), *Quillaja* (an extract of the soapbark tree *Saponaria*), *Purpureocillium lilaciunum* strain 251 (a parasitic fungus), abamectin (from *Streptomyces avermitilus), Burkholderia sp*. strain A396 (a bacteria), and azadirachtin (an extract from the Neem tree) (Westerdahl, 2024).

The utilization of seed-applied nematicides has surged in recent years, emerging as a prevalent method for nematicide application in row-crop agriculture. Within the realm of seed-applied nematicides, seeds treated with biological nematicides either *Pasteuria nishizawae*-Pn1 or Abamectin along with an insecticide and multiple fungicides are available for crops including corn, cotton, and soybeans (Gaspar et al., 2014).

13.10 Cultural Control

A range of cultural practices can be employed for management of nematodes and diseases. These practices encompass a diverse set of strategies such as crop rotation, planting resistant varieties, fallowing, using cover crops, carefully determining dates for planting and harvesting, inducing flooding, utilizing trap crops, promptly removing plants showing symptoms, incorporating soil amendments, ensuring proper plant nutrition and watering, maintaining proper plant spacing and good air circulation, pruning infected plant parts to reduce disease spread, disposing of infected plant material properly, regularly inspecting plants for early signs of disease, and monitoring environmental conditions that may promote disease development.

13.10.1 Crop Rotation

Though crop rotation may initially seem like a straightforward approach to nematode management, the process of implementing a crop rotation plan reveals that several critical aspects need to be carefully considered. Key factors to take into account include the types of nematodes and pathogens existing in the field, the host range of these species, potential rotation crops, anticipated rates of population growth and decline, availability of resistant crop varieties, timing of planting and harvesting, damage thresholds, and the impact of weeds. Once the species in the field are identified, one can refer to host lists or databases to determine the availability of nonhost crops that could be economically viable or seek out suitable resistant crop varieties.

Additionally, consulting these resources may reveal whether local weed species serve as hosts for pests found in the field (Brown, 1987; Ferris, 2024; Roberts, 1993).

Crop rotation is commonly employed for annual crops or long-term perennials like alfalfa that can be cultivated profitably for several years before being rotated to a different crop. Similarly, producers of enduring perennials such as grapes, stone fruits, and nuts should also consider long-range planning. In situations where perennial crops are infested, the likelihood of needing earlier replanting compared to non-infested areas is high. Opting for an interim period of cultivating nonhost annual crops for several years may help circumvent the necessity for chemical interventions, a particularly crucial consideration given the increasingly limited availability of fumigants for preplant use. Choosing annual crops allows sufficient time for the decomposition of woody roots, thereby expediting the decline of pest populations (McKenry et al., 1994; Westerdahl et al., 1998).

The rate of population growth on a host crop is contingent upon factors such as the species present, the specific host crop being cultivated (including different varieties), and various environmental conditions like soil temperature, type, and moisture content. In many cropping systems, there is typically one primary high-yielding crop that generates the greatest returns for growers. As such, a key strategy in crop rotation programs is to maximize the planting frequency of this economically important crop.

Anticipating several years ahead is advantageous in a crop rotation plan. Having a clear understanding of the anticipated rate of nematode and pathogen population growth enables growers to project population levels at harvest. Coupled with knowledge of the expected decline rate, this foresight facilitates planning for the optimal timing of replanting the most profitable crop in the rotation. Additionally, alternative strategies such as early harvesting to potentially reduce nematode and pathogen numbers at harvest and possibly shorten the rotation cycle can also be considered for maximizing efficiency (Smiley et al., 1994).

Crop rotation programs rely on the principle that nematodes and pathogens will gradually perish due to starvation in nonhost environments. The duration for nematodes to starve varies across different nematode species, with limited research conducted on this aspect for many species. The rate of decline to non-damaging levels varies among nematode types. Root-knot nematodes may reach non-damaging levels in 1 to 2 years, cyst nematodes require 3 to 8 years, stem and bulb nematodes take about 4 years, and lesion nematodes may need four years or more. Understanding the rate of population decline is crucial for estimating the requisite number of years for a successful rotation strategy (Ferris, 2024).

13.10.2 Roguing

The technique of removing and managing infested plants in localized areas of a field can be a practical approach when nematode and disease populations are not widespread. For example, early identification of distinctive symptoms in host plants affected by nematodes can enable growers to take targeted action to prevent the spread of infestation to surrounding vegetation. For instance, in the case of daffodils infested with stem and bulb nematode (*Ditylenchus dipsaci*), characteristic symptoms such as leaf markings known as spikkles with raised bumps and yellowing can serve as early indicators of nematode presence. Similarly, on alfalfa, this same nematode may induce symptoms like the shortening of internodes in affected plants, contrasting with the healthy appearance of unaffected alfalfa.

By promptly recognizing these distinct symptoms and being aware of the nematode species causing them, growers can strategically identify and remove infested plants within a field. This targeted removal of infested vegetation can help contain and limit the spread of nematodes to adjacent plants, reducing the potential for widespread infestation and mitigating detrimental impacts on crop productivity (Ferris, 2024).

13.10.3 Resistance

The utilization of resistant plant varieties has been a valuable tool in effectively managing plant parasitic nematodes in agricultural cropping systems. Resistant cultivars offer a sustainable and environmentally friendly approach to nematode control by reducing nematode populations and minimizing crop damage. Various crop species have been developed with resistance to specific nematode species, such as root-knot nematodes, across a range of agricultural commodities including tomatoes, alfalfa, tobacco, grapevines, fruit tree rootstocks, soybeans, lima beans, cotton, sweet potatoes, and coffee (Roberts, 1992).

When considering the use of resistant varieties, it is crucial to inquire about three key aspects: (1) the specific nematode species the variety is resistant to, (2) whether nematodes can cause damage to the variety, and (3) if nematodes can reproduce on the variety. Plants that are immune to nematode attack prevent any form of invasion by the nematodes, including initial root penetration. In contrast, resistant or nonhost plants may still be invaded by nematodes, leading to observable damage; however, the plant's chemical or physical characteristics make it unsuitable for significant nematode population growth. Susceptible plants, on the other hand, facilitate normal nematode reproduction and may or may not exhibit tolerance to nematode attacks (Anand, Koenning and Sharma, 1995; Roberts, 2002).

For example, nematode-resistant tomatoes are typically resistant to three out of the four major nematode species found in tomato-growing regions, with susceptibility remaining to *M. hapla* and possibly other species. In addition to nematodes, VFN tomatoes have resistance to *Verticillium* and *Fusarium*. While resistant varieties offer valuable protection, it is not advisable to repeatedly plant them in the same field year after year. Root knot nematode populations have the capacity to develop resistance to host plant resistance mechanisms when they are consistently exposed to varieties sharing the same genetic background. Therefore, rotating different varieties and employing diverse management strategies are essential for maintaining long-term nematode control efficacy (Hasan and Khan, 1985; Kaloshian et al., 1996).

Research has been conducted illustrating that nematode-resistant tomatoes, when grown in rotation with cotton or beans, can offer a viable alternative to chemical control methods in certain cases. The inability of root-knot nematodes to reproduce on resistant tomato plants results in a scenario where planting these resistant varieties is akin to keeping the land fallow for a period, effectively causing the nematodes to diminish in numbers due to lack of suitable hosts. In a cotton experiment, following a susceptible tomato crop, a comparison was made between growing susceptible tomatoes post-fumigation and resistant tomatoes without fumigation. The results indicated that nematode populations after the resistant tomato cycle were significantly lower, measuring less than half of those observed after the fumigation/susceptible tomato cycle at harvest. In the subsequent year of the experiment, cotton was cultivated both with and without fumigation following the two earlier treatments. Yields across all treatments were found to be comparable, with the exception of cotton grown without fumigation after the fumigation/susceptible tomato cycle, showing a distinct yield disparity. These findings underscore the potential benefits of incorporating nematode-resistant crops in crop rotation strategies as a sustainable approach to nematode management, reducing reliance on chemical interventions while maintaining crop productivity (Roberts and May, 1986).

In another scenario where root-knot nematode-resistant tomatoes were employed to lessen the requirement for chemical control in a subsequent crop, the sequence of crops was as follows: susceptible tomatoes were initially cultivated with fumigation in the first year. Subsequently, in the following year, resistant tomatoes were planted without fumigation. This was succeeded by a cycle of susceptible bean crop cultivation. The outcome of the experiment indicated that the yield of beans remained consistent regardless of whether the field had been subjected to fumigation. This suggests that the utilization of nematode-resistant tomatoes in the crop rotation sequence effectively mitigated the nematode population to a level where the yield of the subsequent susceptible bean crop was not significantly affected (Roberts and May, 1986).

Utilizing resistant plant varieties has proven to be an effective strategy in managing specific nematode species, such as soybean cyst nematode (*Heterodera glycines*) in soybeans and sugarbeet cyst nematode (*Heterodera schachtii*) in sugar beets. The presence of sugarbeet cyst nematodes has been documented in over 40 countries. Over the years, breeding programs have been actively engaged in developing sugar beet varieties resistant to cyst nematodes to mitigate the economic impact of nematode infestations on sugar beet production. It is important to note that while nematode-resistant commercial varieties have shown enhanced performance and yield benefits in the presence of high nematode populations, their performance may vary under different nematode pressure levels. Factors such as nematode population density and environmental conditions can influence the effectiveness of nematode-resistant varieties in achieving yield improvements (Kaloshian et al., 1996; Ogallo. et al., 1999).

A single variety may offer resistance to only one particular nematode genus while remaining vulnerable to others. For example, Nemaguard rootstock, known for its resistance to root-knot nematodes, may paradoxically be more susceptible to damage caused by other nematode species, such as ring nematodes, in comparison to alternative rootstocks like Lovell (McKenry et al., 1994).

An extensive breeding program to develop grape rootstocks has resulted in a number of releases with resistance to multiple species of nematodes and to *Phylloxera* (Table 13.1). Breeding programs are actively working towards developing additional nematode-resistant (or tolerant) varieties for various crops such as carrots, beans, tomatoes, wheat, grapes, as well as fruit and nut trees (Ferris, Zheng, and Walker, 2021; Foundation Plant Services, 2024; Gu and Ramming, 2005).

Table 13.1 Resistance levels of grape rootstocks released by three breeding programs. The first rootstock with resistance to the grapevine fanleaf virus vector *X. index* was O39-16 released in 1991. RS-3 and RS-9 rootstocks susceptible to *X. index* but with resistance to multiple species of root-knot nematode plus ring, lesion and citrus nematode were released in 2003. In 2008, the UCDGRN series with resistance to *X. index* plus additional species were released. This was followed by release of the USDA series, also with resistance to *X. index* and multiple other nematodes (author's own table developed from data in Ferris, Zheng, and Walker, 2021; Foundation Plant Services, 2024; Gu and Ramming, 2005).

Rootstock[a]	*Cx*[b]	*Pv*	*Ts*	*Xi*	*Xa*	*Mi*	*Ma*	*Mih*	*Mj*	*Mc*
O39-16	S[c]	S	S	R						
RS-3	MR	R	MR	S		R	R	R	R	MR
RS-9	MR	R	MR	S		R	R	R	R	
UCDGRN1	R	R	R	R		R	R	R		
UCDGRN2	MR	MR	MS	R		R	R	R		
UCDGRN3	MS	MR	MS	R		R	R	R		
UCDGRN4	MR	MR	MR	R		R	R	R		
UCDGRN5	MS	MS	R	R		R	R	R		
USDA 10-17A	MS	R	R	R		MR			R	MR
USDA 10-23B	MR	R	R	R		R			R	R
USDA 6-19B	MR	R	R	MR	R	R			R	MR

[a]Rootstock Breeding program:

039-16 – Released in 1991 by H. P. Olmo

RS-3, RS-9 – Released in 2003 by D. Ramming and M. V. McKenry

UCDGRN1, UCDGRN2, UCDGRN3, UCDGRN4, UCDGRN5 – Released in 2008 by M. A. Walker

USDA 10-17A, USDA 10-23B, USDA 6-19B – Released in 2012 (10-17A) by USDA

[b]Nematodes:

Cx – *Criconemoides xenoplax* (ring)

Pv – *Pratylenchus vulnus* (lesion)

Ts – *Tylenchulus semipenetrans* (citrus)

Xi – *Xiphinema index* (dagger, transmits grapevine fanleaf virus)

Xa – *Xiphinema americanum* (dagger, transmits virus)

Mi – *Meloidogyne incognita* Race 3 (southern or cotton root-knot)

Ma – *Meloidogyne arenaria* virulent on Harmony rootstock (peanut root-knot)

Mia – *Meloidogyne incognita* virulent on Harmony rootstock (southern root-knot)

Mj – *Meloidogyne javanica* (Javanese root-knot)

Mc – *Meloidogyne chitwoodi* (Columbia root-knot)

[c]Resistance level:

R – Resistant

MR – Moderately Resistant

S – Susceptible

13.10.4 Grafting of Annual Crops

Grafting refers to the intentional fusion of a scion and a rootstock, sourced from different yet compatible plants within close taxonomic proximity, with the aim of creating a composite plant. The scion, which constitutes the upper section, is typically chosen for its favorable characteristics, such as increased yields, larger fruit sizes, or superior flavor profiles. The practice of grafting vegetables can be traced back to its origins in Japan and Korea during the late 1920s. The earliest documented example of interspecific grafting for enhanced productivity and pest and disease management involved watermelons (*Citrullus lanatus*) as the scion grafted onto squash (*Cucurbita moschata*). The watermelon grafting technique was subsequently disseminated among farmers in Japan and Korea between the 1920s and 1930s, eventually expanding to include other vegetable crops such as cucumber (*Cucumis sativus*) and eggplant (*Solanum melongena*) in the 1950s, and later tomatoes (*Lycopersicon esculentum*) (Louws, Rivard, and Kubota, 2010; Sakata, Ohara, and Sugiyama, 2008; Thies et al., 2010).

The primary objectives of vegetable grafting include conferring resistance to soilborne pathogens, as well as boosting yields and enhancing resilience to adverse environmental conditions. Enhanced resistance to soilborne diseases stands out as a key motivation for the adoption of vegetable grafting. Rootstocks are chosen based on their ability to withstand prevalent diseases affecting vegetable crops, including those caused by *Verticillium*, *Phytophthora*, *Fusarium*, and nematodes. Studies have demonstrated that vegetable grafting can significantly increase fruit yields of crops like tomatoes and eggplants, while also improving nutrient absorption and water use efficiency. An enhanced water use efficiency and nutrient uptake capability in grafted plants not only equips them to endure brief dry periods but also boosts their photosynthetic activity (Thies et al., 2010).

Grafting watermelons has emerged as a valuable technique that offers multiple benefits including disease resistance, improved tolerance to adverse environmental conditions, and increased yields. Primarily, two types of plants are commonly employed as rootstocks for grafting watermelons: bottle gourd (*Lagenaria siceraria*) and interspecific winter squash hybrids (*Cucurbita maxima* x *Cucurbita moschata*). Additionally, grafting watermelons is now being recognized for its utility in enhancing tolerance to extreme temperatures, optimizing nutrient absorption, increasing water use efficiency, mitigating the effects of alkalinity, salinity, and flooding, countering mineral toxicities, and enhancing overall yield, quality, and size of fruits. Bottle gourd rootstocks are known for their resistance to *Fusarium* wilt and ability to withstand chilling conditions. While the root growth of bottle gourd rootstocks may not be as robust as that of interspecific hybrids, they exhibit early maturity. Grafting onto bottle gourd typically does not adversely affect flowering or fruit quality. Studies illustrate that grafting can effectively address a plethora of soilborne diseases impacting watermelon, including those stemming from fungi, bacteria, and nematodes. Some rootstocks may even offer resistance against foliar diseases. Furthermore, grafting proves beneficial in managing various other soilborne diseases such as P*hytophthora* root and crown rot as well as *Verticillium* wilt (Keinath and Hassell, 2014).

Grafted watermelon seedlings come at a higher cost compared to non-grafted seedlings, as seen in a 2014 study where the production cost of grafted seedlings was three times that of non-grafted seedlings. Additionally, the production of grafted seedlings requires specialized equipment, facilities, and skilled labor. Despite the increased expenses, the potential benefits in terms of yield enhancement and Fusarium wilt resistance often justify the higher cost (Louws, Rivard, and Kubota, 2010).

Furthermore, research focusing on secondary plant metabolites such as anti-nematode phytochemicals (ANPs) and the mechanisms underlying plant resistance to nematodes holds promise for developing novel nematode management strategies. By exploring the bioactive compounds produced by plants and understanding the genetic and biochemical pathways involved in nematode resistance, researchers can uncover new avenues for enhancing crop resistance to nematodes and developing innovative approaches for integrated nematode management in agriculture.

Overall, the incorporation of resistant plant varieties, coupled with advancements in understanding plant-nematode and plant-disease interactions at the molecular level, presents opportunities for improving nematode and plant disease control strategies and promoting sustainable agricultural practices that reduce reliance on chemical nematicides while enhancing crop productivity and soil health.

13.10.5 Screening Available Cultivars

In the case of managing the Columbia root-knot nematode, extensive screening of potato varieties was carried out. While no completely resistant varieties were identified for use in the Tulelake/Klamath Basin of California, the research did reveal that certain potato varieties exhibited reduced blemishing compared to the standard variety, Russet Burbank. This indicates the potential for selecting varieties that may not be nonhosts but are less susceptible to nematode damage, thereby reducing economic losses and production challenges (Carlson, Westerdahl, and Ferris, 1992).

Similarly, field screening of wheat and barley varieties, which are commonly rotated with susceptible crops, also proved beneficial. Varietal screening showed that some wheat and barley varieties were less favorable hosts for nematode reproduction compared to others. Steptoe and Briggs barley varieties were found to support lower nematode reproduction rates than Fieldwin wheat or Crystal barley, indicating their potential as better rotation options to manage nematode populations in the soil (Carlson, Westerdahl, and Ferris, 1992).

Such research findings emphasize the importance of varietal selection in nematode and disease management strategies and highlight the value of incorporating tolerant or less susceptible crop varieties in agricultural practices to mitigate nematode damage and maintain sustainable crop production systems.

13.10.6 Cover Crops

Cover crops can improve soil fertility by adding organic matter when they decompose, enhancing soil structure, and promoting microbial activity. Certain cover crops can scavenge and store nitrogen, reducing leaching and making it available for subsequent cash crops. Cover crops help reduce erosion and can compete with weeds for resources such as light, water, and nutrients, thereby reducing weed pressure. Understanding the interactions between cover crops, nematodes and plant pathogens is a critical aspect of cultural management in agricultural systems. Currently, our knowledge of cover crops is primarily focused on predicting which cover crops are less likely to worsen existing issues rather than actively reducing populations beyond what occurs in fallow conditions. Selecting an appropriate cover crop involves considering the nematode and plant pathogen species present in a specific field. The diversity of nematode and pathogen species that may coexist in the soil, make it challenging to identify a single cover crop that effectively suppresses species of interest. Since nematodes and pathogens are typically plant stressors, any practices that mitigate stress from other sources can indirectly benefit crop health in infested environments (Caswell-Chen and Goodell, 1992; Ferris, 2024).

The utilization of cover crops in the management of sugar beet cyst nematodes represents a promising development in nematode control strategies. Certain cover crops, such as mustard, oil radish, and buckwheat, which are known hosts of sugar beet cyst nematodes, have shown effectiveness in reducing nematode populations and altering their life cycle dynamics. Notably, breeders in Germany have identified specific varieties of these host cover crops that stimulate egg hatch and may even facilitate nematode penetration of roots. This selective breeding has led to significant reductions in nematode populations, with reported decreases of up to 50% observed upon planting cover crops. Additionally, the shift in nematode development from females to males or the prevention of regular development indicates a disruption in the nematode life cycle, potentially inhibiting their reproductive capability. While certain cover crops have shown promise in Germany and other regions, it is important to consider regional variations in nematode populations and crop interactions. In California, where different populations of sugar beet cyst nematodes exist, various cover crop varieties have been tested and shown to be hosts (Gardner and Caswell-Chen, 1993).

13.10.7 Fallow

Fallowing, or leaving the soil without a host crop, is a traditional method used to manage nematodes by depriving them of a host plant, ultimately leading to their decline or death due to starvation. This approach is based on the principle that nematodes cannot survive indefinitely without a suitable host (Duncan, 1986).

For certain economically important nematodes like root-knot, sugarbeet cyst, stem and bulb, and *X. index*, there is existing knowledge regarding the duration nematodes can persist without a host before their populations decline. However, for many other nematode species, this critical information is still lacking, making it challenging to effectively implement fallow as a nematode management strategy (Johnson, et al., 1992).

One common limitation in utilizing fallow effectively is the oversight of weed hosts that can serve as alternative hosts for nematodes during the fallow period. Weeds can sustain nematode populations, undermining the efficacy of fallow in reducing nematode numbers. Additionally, the persistence of root remnants in the soil can continue to serve as hosts for nematodes even in the absence of above-ground plant growth. The effectiveness of dry versus wet fallow in managing nematodes remains a topic of debate. The efficacy of either dry or wet fallow can be influenced by various factors such as the specific nematode species present, soil type, climate conditions, and other environmental variables (Goodell and Ferris, 1989; Smiley et al., 1994).

Studies suggest that soil disturbance practices like plowing or rototilling during fallow periods can expedite the decline of nematode populations. However, while soil disturbance may enhance nematode reduction, it can also increase the overall cost of fallow procedures, adding to the economic considerations that growers must take into account (Johnson and Motsinger, 1990; Minton, 1986).

13.10.8 Flooding

When considering the use of flooding as a potential method to control nematodes, it is important to recognize key factors that influence its effectiveness. Soil-dwelling nematodes, being aquatic organisms that reside in the thin film of moisture surrounding soil particles, possess adaptations that can limit the efficacy of flooding as a nematode management strategy. Additionally, some nematodes have the capability of anaerobic respiration, which allows them to survive under waterlogged conditions, further complicating the control of nematode populations through flooding (Sikora, Bridge, and Starr, 2005).

However, it is essential to note that while continuous flooding may not be the most effective strategy for nematode management in certain cases, flooding can still play a role in nematode control under specific conditions. For instance, in situations where flooding is implemented for purposes such as salt leaching or waterfowl management, incidental reductions in nematode populations can also be realized. This highlights the potential for integrating nematode management objectives with other agricultural practices that involve flooding.

Moreover, in different nematode species and geographical locations, short-term flooding has been utilized as a strategy to stimulate nematode egg hatch. Subsequent fallowing following this inundation can lead to a more rapid decline in nematode populations. This approach leverages flooding to disrupt nematode life cycles and create conditions that are less hospitable for their survival, ultimately aiding in population reduction (Sikora, Bridge, and Starr, 2005).

13.10.9 Predictive Models

Through the application of predictive models like the Seinhorst Model and its variations, growers can enhance their ability to anticipate and manage nematode infestations, thereby optimizing crop production and sustaining agricultural productivity in the presence of nematode pressures. The Seinhorst Model, developed in 1965, has served as a foundational concept for various subsequent models aimed at predicting and managing crop damage resulting from nematode infestations. This model establishes a quantitative functional relationship between nematode population density at planting and relative crop yield in annual crops. Understanding this relationship could help growers make informed decisions about nematode management strategies to mitigate yield losses effectively.

In the Seinhorst Model, the relationship between nematode population density and crop loss is expressed by the following equation:

$Y = M / (1 + (P / T)^Z)$

Where:

- Y represents crop loss,
- M denotes the minimum relative yield achievable in the presence of high nematode densities,
- T signifies the initial nematode density at which relative yield starts to decline (referred to as the tolerance limit),

- Z is a parameter reflecting the per capita damage to plant roots caused by nematodes, often representing the proportion of the root system that remains undamaged by a single nematode (typically slightly less than one),

- P represents the initial nematode density before planting (also denoted as Pi) (Duncan and Ferris, 1983; Seinhorst, 1965).

13.10.10 Damage and Economic Thresholds

In certain pest management fields, such as entomology, the concept of utilizing damage thresholds and economic injury levels is commonplace. This familiarity can sometimes lead to the presumption that this approach is equally applicable to other areas within pest management. While the concept of damage thresholds holds merit for nematodes and plant pathogens, the actual development and utilization of specific damage thresholds have proven to be challenging. In many nematode-crop associations, the establishment of precise damage thresholds remains elusive. Despite the concept's theoretical validity, practical implementation has encountered obstacles in the realm of nematology, with gaps existing in the availability of damage thresholds for various nematode species affecting crops (Abawi and Barker, 1984; Ferris, et al., 1986; McSorley and Duncan, 1994).

As an example, when fields are sampled at harvest in late summer or fall, population levels in infested fields are readily detectable. Following this, if fields are left fallow or planted with a nonhost crop during the winter, the observed rate of population decline is approximately 85 percent. As a result, nematode populations in the spring are significantly reduced, with levels often dropping below detectable limits using standard extraction methods. If subsequently, a susceptible crop like tomatoes is planted, nematode populations gradually increase until harvest (Table 13.2). Expected yields based on the initial nematode populations within the field can be estimated. To establish meaningful population thresholds for nematodes in annual crops, it appears essential to base assessments on samples taken at harvest for subsequent crops rather than shortly before planting, which is the conventional practice. This strategic shift in sampling timing can provide more accurate data for managing nematode populations effectively and optimizing crop yields in the long term (Flint, 1998).

Table 13.2 An illustration of the relationship between initial populations of root-knot nematode and the expected yield of processing tomatoes in the San Joaquin Valley of California, USA. As described by Flint (1998) from the time that tomatoes are harvested in the fall, until tomatoes are planted again in the spring, populations are expected to decline by 85%. The expected increase in nematodes during the growing season is related to the size of the initial population. At lower initial populations, the root system can support a greater rate of reproduction than it can at higher initial populations (author's own table).

Fall population[a]	Percent decline	Spring population	Increase	Fall population	% of normal yield
		0.01	1000 X	10.00	100
0.31	85	0.05	500 X	23.80	98
1.56	85	0.25	150 X	37.30	85
4.06	85	0.65	75 X	48.00	65
6.25	85	1.00	55 X	54.80	53

[a]Juvenile nematodes/gram of soil

Damage thresholds are commonly perceived as fixed numerical values. However, for nematodes, environmental factors such as temperature at planting time play a significant role in determining the extent of potential damage. If the temperature at planting falls below the activity or infectivity threshold specific to the nematode species in question, the resultant damage is typically reduced compared to instances with warmer temperatures. *Pratylenchus thornei* is known to be a significant issue for wheat crops in Mexico. Despite being prevalent in fields in northern California, this nematode species does not pose a threat to wheat crops in the region. This contrast is likely attributed to the cultivation of wheat during the winter months when soil temperatures remain below the nematode's activity threshold (VanGundy et al., 1974).

13.10.11 Date of Planting

Temperature plays a major role in the rate of development of nematodes and plant pathogens. The relationship between soil temperature and plant susceptibility underscores the significance of considering temperature dynamics in management strategies. Each nematode species has a minimum temperature threshold below which they are unable to effectively penetrate plant roots, which can impact their ability to cause damage. Planting crops during periods when soil temperatures are below the infection minimum can help mitigate nematode damage by limiting their activity (Jeffers and Roberts, 1993; Johnson and Motsinger, 1990; Roberts, 1987).

Manipulating planting dates based on temperature considerations can reduce nematode damage on crops like carrots and wheat. By aligning planting schedules with optimal soil temperatures that deter nematode penetration, growers can potentially minimize nematode-induced losses. For instance, studies have shown that adjusting planting dates by a few weeks to coincide with temperatures unfavorable for nematode activity can lead to significant reductions in nematode damage, as illustrated by a graph depicting soil temperature variations throughout the year relative to nematode infection thresholds (Fig. 13. 2) (Roberts, 1987; Roberts, VanGundy, and McKinney, 1981).

Fig. 13.2 The relationship between soil temperatures and the nematode activity threshold is evident in the case of *M. incognita*, which cannot penetrate roots when soil temperatures fall below 18°C. Shifting the planting schedule from mid-October to mid-December enables crop cultivation without incurring nematode damage. Daily maximum and minimum soil temperature data for Orange County, CA was obtained from UC IPM Online (2024c) and graphed following a method described by Roberts (1987).

13.10.12 Date of Harvest

Adjusting the timing of harvest can be a valuable strategy in managing nematode damage in certain situations. By strategically planning the harvest timing based on predictions of nematode population dynamics and economic considerations, growers can potentially minimize the impact of nematodes on crop yield and quality. Effective utilization of this technique requires the ability to forecast when nematode populations will reach economically significant levels, prompting decisions for an early harvest to mitigate damage (Carlson, Westerdahl, and Ferris, 1992, Pinkerton, Santo, and Mojtahedi, 1991).

In the field of entomology, the concept of degree days or heat units has proven to be a valuable tool for predicting the population cycling of pest insects. This methodology calculates the accumulation of heat units over time to estimate the developmental rates of insect populations and predict key life cycle events. Sufficient information is available for root-knot nematode, for sugar beet cyst nematode, and several fungal diseases to implement degree day models. By tracking nematode degree days and monitoring their population dynamics in relation to crop growth stages, growers can make informed decisions regarding harvest timing to minimize nematode damage. Further research and development in nematode degree day modeling could expand the applicability of this approach to a wider range of nematode species and cropping systems, offering growers additional tools for sustainable nematode management (Ferris and VanGundy, 1979; Noling and Ferris, 1987; Pinkerton, Santo, and Mojtahedi, 2009; Schneider and Ferris, 1987; UC IPM Online, 2024).

13.10.13 Trap Cropping

Trap cropping can be highly effective strategy in nematode management due to the specific mechanism through which it targets and controls nematode populations. When an infective juvenile nematode penetrates a host root and begins feeding, it undergoes physiological changes, becoming swollen or rotund, ultimately becoming trapped within the root tissues. By destroying the host plant before nematodes can lay eggs, the trapped nematodes within the roots perish, thereby reducing the nematode population in the soil. One key aspect to note is that trap cropping is generally not effective for managing migratory ectoparasitic or endoparasitic nematodes. This limitation stems from the ability of these nematode species to move between different plant roots and continue their feeding and reproductive activities (Gardner and Caswell-Chen, 1997; Scholte, 2000).

Trap cropping is another technique that relies on temperature monitoring. It can be employed for reducing populations of sedentary endoparasitic nematodes like root-knot (*Meloidogyne* sp.) or cyst nematodes (*Heterodera* sp.). This method involves planting a host crop that attracts the nematodes of interest, allowing it to grow for a period, and then terminating it before the development of egg-bearing adults. It is crucial to ensure that the roots of the trap crop are thoroughly destroyed to prevent them from serving as a continued food source for nematode development. The success of trap cropping hinges on the precise timing of crop termination. Nematodes develop more rapidly in warmer soil compared to cooler soil conditions. Therefore, in regions with warmer climates, the termination of the trap crop needs to occur earlier to effectively control nematode populations, compared to regions with cooler climates. Understanding the heat unit requirements, measured as nematode degree days, for the development of different species of root-knot nematodes is crucial for determining the optimal timing for crop termination. Different nematode species have varying heat unit requirements for development, emphasizing the necessity of identifying the specific nematode species present in a field for successful nematode management (Noling and Ferris, 1987; UCIPM, 2024b)

In the case of trap cropping, key cost components include irrigation for growing the trap crop or germinating nematode-susceptible weeds, the expenses related to acquiring trap crop seeds, planting operations, and the costs associated with crop termination. These factors contribute to the overall financial investment required for implementing trap cropping as a nematode management tactic. When comparing the cost of irrigation for trap cropping with the expenses associated with traditional chemical control methods, particularly fumigant nematicides commonly used on vegetable crops, it is notable that both approaches require irrigation. For instance, chemical control methodologies often necessitate water applications for applying fumigants like metam sodium products or for soil sealing to reduce emissions. In scenarios where weed species present in the field serve as hosts for the nematodes targeted for control, these weeds can potentially function as natural trap crops. By allowing these weeds to germinate and strategically terminating them before seed development, growers can leverage weed management as an integral part of their nematode control strategy, thereby maximizing cost-efficiency (Westerdahl, 2020, 2021).

13.10.14 Biofumigation

Research efforts focusing on biofumigation as a sustainable approach for managing nematodes, weeds, and fungi have provided valuable insights into utilizing *Brassica* species, such as broccoli, for nematode control through the release of glucosinolates and subsequent production of isothiocyanates in the soil during degradation. *Brassica* plants contain glucosinolates, which are secondary metabolites known for their biofumigant properties. When

Brassica residues are tilled into the soil, glucosinolates break down and release isothiocyanates, which possess nematicidal properties. Intriguingly, metam sodium, a conventional nematicide, also employs isothiocyanate compounds as its active ingredient for nematode control (Kirkegaard et al., 1998).

In soil fumigation practices, plastic film tarping is commonly used to reduce the volatilization rate of fumigants applied to the soil. Similarly, when incorporating biofumigation techniques in nematode management, utilizing plastic film coverings over the soil can enhance the effectiveness of biofumigants. This tarping technique helps to contain and concentrate the released isothiocyanates within the soil profile, prolonging their exposure to nematodes and enhancing the overall efficacy of biofumigation for nematode and disease control (Angus et al., 1994; Gamliel and Stapleton, 1993; McFadden, Potter, and Brandle, 1992; Mojtahedi et al., 1991; Ploeg, 2007; Ploeg and Stapleton 2001; Spak et al., 1993; Stapleton, Duncan, and Johnson, 1998).

13.10.15 Anaerobic Soil Disinfestation (ASD)

The ASD method offers a promising approach for managing nematodes, pathogens and weeds. The process involves the application of powdery rice bran to initiate the ASD process. Subsequently, the bran is incorporated into the soil, followed by the application of a clear tarp and auxiliary irrigation system. During the initial one to two days, the entire system is irrigated to a depth of 1.5 to 2 meters, ensuring thorough saturation of the targeted soil profile. Daily or alternate-day pulsing of the irrigation system thereafter is essential to maintain soil moisture at levels exceeding field capacity. The final stages encompass the removal of the tarp and irrigation infrastructure, preparing the soil for subsequent planting activities (Shennan et al., 2018).

Optimal conditions for ASD application revolve around the availability of ample solar radiation, ideally during the peak of summer or alternatively towards the late summer and early fall seasons. The biological and chemical transformations induced in ASD-treated soils are integral to its efficacy. Through the combined actions of elevated soil temperatures facilitated by the tarp and the saturation of moisture, soil bacteria metabolize the carbon-rich substrate, leading to the generation of organic acids and a reduction in pH levels. These processes result in the release of volatile compounds that collectively modulate the soil microbial community towards a more conducive environment for almond tree cultivation (Shennan et al., 2018).

13.11 Chemical Control

Chemical disease control primarily involves the application of fungicides, bactericides, and nematicides to combat biotic agents causing plant diseases. Pesticides play a significant role in managing certain disease scenarios where their use is indispensable and effective.

13.11.1 Fumigants

The fumigant carbon disulfide was initially trialed in Europe for combating *Phylloxera* infestations on grapevines. With its wide-ranging effectiveness, researchers discovered its efficacy against nematodes and certain plant pathogenic fungi. Professionals in nematology who have employed this compound often highlight its flammability, recounting alarming incidents where fields and machinery were consumed by fire if the applicator inadvertently struck a rock during its use. Following World War I, a substantial quantity of chloropicrin remained in the Hawaiian Islands. The pineapple industry there actively backed extensive research endeavors focused on nematode and fungal control. Through these efforts, it was discovered that chloropicrin exhibited both nematicidal and fungicidal properties (Johnson and Feldmesser, 1987).

The successful utilization of these initial fumigants catalyzed the emergence of various other products in the 1940s. These included methyl bromide, ethylene dibromide (EDB), dibromochloropropane (DBCP), D-D Mixture, 1,3-dichloropropene (marketed as Telone and serving as one of the constituents of the D-D Mixture), and 1,2-dichloropropane - the other constituent of the D-D Mixture. During the 1960s and 1970s, the utilization of EDB, D-D, and DBCP was discontinued due to concerns regarding the potential for groundwater contamination and/or carcinogenic effects. The use of methyl bromide is being phased out owing to concerns surrounding its impact on the ozone layer. Ironically, none of these issues were evident during the initial stages of development (Ferguson and Padula, 1994; Heald, 1987; Thomason, 1987).

Metam-sodium, which upon soil application releases methyl isothiocyanate which is a potent broad-spectrum biocide was initially researched as a nematicide in the 1950s. However, due to its performance and lack of ease of use at the time, it struggled to compete with other available fumigants. During the 1980s, sodium tetrathiocarbonate a liquid that decomposes in the soil to release carbon disulfide, was introduced (Johnson, 1985).

Fumigants, aptly named for their high volatility, have the capability to travel through air in soil pores over varying distances (ranging from cm to meters), driven by their volatility before dissolving into the water film around soil particles where nematodes are present. Among the fumigants, metam-sodium exhibits the least volatility and typically moves around 10 cm within the soil. To optimize its efficacy, as well as that of organophosphates and carbamates, these chemicals must be moved through the soil using methods such as incorporating them into irrigation water or employing mechanical tillage (Heald, 1987).

The efficacy of fumigants is influenced by factors such as soil type, temperature, and moisture levels, all of which impact the movement of the product through the air in soil pore spaces. When temperatures are too low, fumigants may have limited mobility through the air-filled pores in the soil. Conversely, at elevated temperatures, the fumigant can rapidly volatilize out of the soil, reducing its effectiveness. Higher soil moisture content can impede the movement of fumigants, as water in the soil pores hinders their diffusion. Additionally, soils with finer textures, characterized by increased silt and clay content, contain smaller pores, which can further restrict the movement of fumigants within the soil matrix (Hague and Gowan, 1987).

Fumigants have historically been applied using shanks, which are knife-like blades. These shanks have a tube that carries the product from the back of each shank to its tip. In traditional fumigation practices, the liquid fumigant is injected below the surface of carefully prepared soil, typically applied in a narrow band as the fumigation equipment traverses the field. To seal or compact the soil surface, a ring roller is often pulled behind the fumigation equipment, or a tarp or irrigation is applied.

Commercial applications of fumigants are typically conducted using sophisticated equipment capable of precise application. This equipment can accurately dispense the product, lay a tarp, and adhere adjacent edges together in a single operation. Despite the apparent complexity of these operations, a more simplified fumigation setup can achieve similar results using a cylinder of methyl bromide pressurized by a smaller nitrogen cylinder, connected to a fumigation shank via a flow meter (Fig 13.3).

Fig 13.3 Equipment used to apply products to soil: upper left - commercial fumigation apparatus, upper right – essential components of a fumigation apparatus (from left to right nitrogen cylinder to pressurize fumigant cylinder, flow meter, fumigation shanks), lower left – tractor drawn sprayer, lower right – tractor mounted granular applicator (author’s own images).

Innovations in fumigation equipment have led to the development of shanks with multiple openings. These modified shanks are particularly useful in soil types where fumigants do not disperse effectively or when using metam-sodium, which has limited movement within the soil. The multiple openings on these shanks enhance the distribution of fumigants throughout the soil, ensuring more uniform coverage and effective pest control (McKenry et al., 1994).

Nematicides will not eradicate nematodes. When applied correctly, they can effectively diminish nematode populations to undetectable levels, enabling the planting of crops and fostering the growth of a robust root system before nematode populations rebound to harmful levels. Effective land preparation is vital for successful fumigation. This process typically involves clearing roots and debris from the previous crop and conducting plowing or subsoiling to a depth of 0.5 to 1 meter to eliminate any restrictive layers within the soil profile. To optimize land preparation, minimizing the presence of organic matter is crucial, with a recommended organic matter content of less than 2%. Key steps for land preparation include cultivating the top 6 to 8 inches of soil is advisable to break up clods that can impede fumigant movement. removing nematode-harboring roots or allowing them to decompose prior to fumigant application can enhance the efficacy of fumigation (Hague and Gowan, 1987).

The rate of fumigant required is influenced by the soil texture, with finer soil textures necessitating higher fumigant rates. Coarse-textured soils, characterized by a greater proportion of sand, typically can be effectively fumigated using lower rates compared to fine-textured soils containing higher proportions of silt and clay. This difference is attributed to the distinct pore space sizes associated with varied soil particle sizes, where fumigants tend to travel more swiftly through larger pores than smaller ones. Broadly speaking, nematicides generally exhibit greater mobility and efficacy in coarse-textured soils compared to fine-textured soils. The ability of fumigants to move more effectively in coarse soils contributes to their perceived higher efficacy in such environments (Hague and Gowan,1987).

Optimum soil temperature ranges are critical considerations for successful fumigation. Interestingly, the optimal temperature range tends to be broader for coarser-textured soils compared to finer-textured soils. This variance is attributed to the different heat retention capacities and thermal properties of soil textures. For coarse-textured soils, the broader optimal temperature range allows for more flexibility in fumigation applications in varying temperature conditions. In contrast, finer-textured soils, with their higher capacity to retain heat, may have a narrower temperature range within which fumigation is most effective (Johnson, 1985).

Optimal soil moisture levels play a crucial role in the efficient dispersal of fumigants. In finer-textured soils, characterized by smaller pore spaces and a higher water-holding capacity compared to coarser soils, managing soil moisture becomes particularly important for effective fumigant application. The presence of excess water in soil pores acts as a barrier to the movement of fumigants in the gaseous phase. Therefore, in water-saturated conditions, fumigants are unable to permeate through the soil effectively. To mitigate this challenge, sealing the soil surface post-application can enhance the control of fumigants in the upper soil layers. This practice slows down the rate at which the fumigant disperses through this region, aiding in the targeted and efficient distribution of the fumigant within the soil profile. Adapting fumigation strategies based on soil moisture conditions and implementing appropriate soil surface sealing techniques are essential steps to optimize fumigant efficacy, especially in finer textured soils where water content can significantly impact fumigant movement and effectiveness (Johnson, 1985).

Whether utilizing elaborate or straightforward apparatus, the fundamental function of a fumigation apparatus remains consistent: to introduce a liquid or gas below the soil surface. This substance disperses in the soil, spreading at least a foot in all directions from the injection point by traveling through the air in soil pores and dissolving in the moisture film surrounding these pores. This dispersion mechanism aims to reduce nematodes and pathogens to nondetectable levels.

13.11.2 Liquid and Granular Products

Fungicides are an important component of plant disease management. Proper fungicide selection hinges on precise diagnosis of the disease issue at hand due to varying efficacy levels of different fungicides. Fungicides are given a FRAC group code based on their mode of action, target site and code, group name, chemical or biological group, common name, and potential for development of resistance. Rotating pesticide classes and adopting resistance management strategies help prevent the development of resistance in pathogens. Protectant fungicides act preventatively and must be applied before pathogen attack to inhibit spore germination or kill spores on the plant surface. Systemic fungicides are absorbed by plants through foliage or roots, systemic fungicides are translocated within the plant. They can act curatively to combat pathogens already present (Agrios, 2005).

During the 1950s and 1960s, several organophosphates and carbamates were developed as nematicides. Liquid products are often applied by spraying them onto the soil surface either before or after planting. For post-plant applications, many growers utilize sprayers commonly used for herbicide application to treat both sides of tree or vine rows effectively (Fig 13.3). Following the application of liquid products, it is essential to incorporate the product into the soil to enhance its efficacy. This incorporation can be achieved through irrigation or the use of mechanical incorporation equipment. Mechanical incorporators come in various types, each offering different depths of product integration into the soil (Johnson, 1985).

The choice of incorporation method depends on factors such as the specific product used, soil type, crop or turfgrass variety, and desired depth of distribution. Both irrigation and mechanical incorporation play crucial roles in ensuring proper dispersion of products within the soil, thereby maximizing their effectiveness in controlling populations and protecting plant health. Growers commonly possess granular applicators designed for the precise application of fertilizers and insecticides (Fig 13.3). These tools can often be adjusted or calibrated to facilitate the accurate application of granular pesticide products as well. Similar to spray applications, when granules are used and applied to the soil surface, it is crucial to promptly incorporate them into the soil for optimal effectiveness. This incorporation can be achieved through irrigation or mechanical devices to ensure proper dispersion and penetration of the granules throughout the root zone where nematodes typically reside.

Drip irrigation offers a range of advantages that contribute to efficient and effective crop management practices. They minimize water loss through evaporation and runoff. Drip irrigation systems are automated and can be programmed to deliver water and nutrients according to specific schedules and plant requirements. Drip irrigation systems can be integrated with pest control products such as nematicides, fungicides, fertilizers, or other agrochemicals. This enables the precise and controlled application of these products directly to the root zone, ensuring efficient utilization and reducing potential waste or environmental impact. The targeted delivery of pest control products through drip irrigation systems results in lower overall usage of these products compared to blanket applications in traditional irrigation methods. Drip irrigation minimizes leaf wetting, which can help reduce the incidence of foliar diseases (McKenry et al., 1994).

The mode of action of carbamates and organophosphates on nematodes is believed to parallel their effects on insects, involving the inhibition of acetylcholinesterase at nerve synapses. In the realm of nematodes, these substances are commonly referred to as "nematistats," Because at lower concentrations, they can impede nematode activity without causing immediate lethality. Moreover, these effects are generally reversible if nematodes are transferred to a water medium. These chemicals do not necessarily kill nematodes outright; instead, they can disorient, paralyze, or confuse the nematodes, effectively preventing root infestation. Over time, prolonged exposure to these compounds can lead to the nematodes starving to death. Organophosphates and carbamates decompose in the soil over a span of several weeks. As a result, there may be a risk of nematodes regaining their ability to feed on and penetrate roots once the chemical presence diminishes (Hough and Thomason, 1975).

In annual cropping scenarios, the primary objective is to achieve nematode control for several weeks to allow a healthy root system to develop and support the crop until harvest. For perennial crops, where nematode pressure persists over longer periods, multiple applications can be strategically timed to provide season long control. By planning and scheduling repeated applications effectively, growers can maintain nematode control and minimize root damage throughout the cropping season in perennial systems (Becker, Ploeg and Nunez, 2019).

The concept of nematistat effects is not universally accepted among nematologists, likely due to varying observations across different cropping contexts. It is possible that the same product may exhibit dual roles as both a nematistat and a nematicide within a specific situation. When considering organophosphates and carbamates, it is crucial to recognize that their effectiveness relies on proper distribution throughout the soil via irrigation water or mechanical incorporation. During application, the distribution of these chemicals may not be uniform, leading to a varied range of concentrations within the soil profile. Consequently, certain areas may experience concentrations high enough to directly kill nematodes, while elsewhere sublethal effects, such as disorientation or paralysis, may be observed. These differential concentrations can result in a complex interplay between nematistatic and nematicidal effects within the same application scenario. Sublethal concentrations of aldicarb were found to suppress the hatch of nematode eggs in some cases, while in other cases, they actually stimulated egg hatch. Furthermore, the exposure also impacted other behaviors of the nematodes. For example, migration, infection, and the attraction of male nematodes to female nematodes could be inhibited under certain concentrations of aldicarb (Hough and Thomason, 1975).

Following the application of pest control products, they can dissipate through various mechanisms. Fumigants, for instance, are formulated to volatilize, potentially leading to their release into the air where they can decompose under sunlight. Within the soil, fumigants can also undergo decomposition via hydrolysis. On the other hand, organophosphates and carbamates typically break down in soil through microbial activity (Johnson and Feldmesser, 1987).

Reports have emerged of a decline in nematicidal efficacy following repeated applications. In some instances, microbial communities capable of decomposing organophosphates and carbamates may become selected for and proliferate over time. This overgrowth can result in accelerated breakdown rates, potentially rendering them ineffective for control purposes (Davis, Johnson, and Wauchope, 1993).

In addition to the biological products mentioned in the section on Biological Control, several additional active ingredients have recently become commercially available or are seeking registration as nematicides including: fluensulfone, fluazaindolizine, fluopyram (initially registered as a fungicide), spirotetramet (initially registered as an insecticide), and allyl isothiocyanate (Becker, Ploeg and Nunez, 2019).

13.12 Examples of Integrated Treatment Programs

13.12.1 Steps in Developing a Treatment Program

Developing an effective treatment program is crucial for managing nematodes and plant diseases and minimizing crop damage in agricultural systems. Key steps involved in the development of a comprehensive treatment program include.:

1) Know the field history: Understanding the historical practices, crop rotations, previous nematode and plant disease issues, and treatments applied in the field provides valuable context for developing a targeted management strategy.

2) Identify the nematodes and pathogens present: Conduct nematode surveys and diagnostic tests to identify the specific nematode species present in the field. Different species require tailored management approaches, so accurate identification is essential.

3) Consider the biology of the nematodes and pathogens present: Understanding the life cycle, behavior, and seasonal dynamics of the species present influences the choice of control methods and timing of interventions.

4) Establish a damage threshold: Determine the threshold population levels at which nematodes and pathogens cause economically significant damage to crops. Establishing a damage threshold helps in making informed decisions on when to implement control measures.

5) Evaluate nonchemical alternatives: Explore and assess nonchemical management strategies, such as crop rotation, resistant cultivars, soil amendments, and biological control agents.

6) For chemical alternatives:

- Soil preparation: Ensure proper soil preparation practices to optimize the efficacy of chemical treatments and enhance root health.

- Comparison areas: Leave untreated control areas within the field to compare the effectiveness of chemical treatments and monitor populations over time.

- Consultation: Seek guidance from agricultural advisors, extension services, and other experts to access the most up-to-date information on management strategies and chemical options.

- Application method and timing: Determine the most appropriate method, application rate, and timing for applying products based on the species, crop stage, soil conditions, and environmental factors (Hirano, 1975; Pitcher, 1978; Powell, 1971).

By following these steps and integrating a combination of cultural, biological, and chemical control measures tailored to the specific species and field conditions, farmers and agronomists can develop a targeted and sustainable treatment program to effectively manage infestations and protect crop yields.

13.12.2 Management of Sugarbeet Cyst Nematode on Sugarbeets

The control and management of sugarbeet cyst nematode on sugarbeets, as outlined in the study by Raski and Allen from 1948, emphasize several key practices to mitigate nematode infestation and reduce crop damage (Raski and Allen, 1948).

Implement measures to prevent the spread of sugarbeet cyst nematode to uninfested areas. This could include using clean machinery and equipment, controlling the movement of infested soil, and maintaining proper sanitation practices to reduce the risk of nematode introduction into new fields.

Manage and control weeds that serve as hosts for sugarbeet cyst nematode. By reducing weed populations that can harbor nematodes, farmers can help limit nematode reproduction and spread in the field.

Utilize crop rotation strategies to disrupt the nematode life cycle and reduce nematode populations in the soil. By planting non-host crops or alternative crops that are less susceptible to sugarbeet cyst nematode, farmers can help manage nematode numbers and decrease the likelihood of crop damage.

Consider planting sugarbeets early in the season when temperatures are still too low for nematode activity or infection. Lower temperatures can inhibit nematode development and reproduction, reducing the impact of sugarbeet cyst nematode on crop yield and quality.

13.12.3 Management of Columbia Root-knot Nematode on Potatoes

Prevention:

- Washing Equipment: Regularly clean and sanitize equipment used in potato production to prevent the spread of nematodes between fields.
- Certified Planting Stock: Start with disease-free and certified planting material to minimize the introduction of Columbia root-knot nematode into new potato fields.

Damage Thresholds:

- Use fall population levels of nematodes to set damage thresholds. Monitoring nematode populations can help determine the need for interventions and control measures.

Crop Rotation:

- Rotate crops with non-host plants such as alfalfa, barley, wheat, onions, and fallow periods to disrupt the nematode life cycle and reduce nematode populations in the soil.

Harvest Dates:

- Determine harvest dates based on accumulated degree days to optimize potato growth and yield while considering nematode activity and impact on the crop.

Chemical Control:

- Consider chemical control methods only in fields with low nematode populations. This targeted approach can help manage nematodes effectively without unnecessary use of chemical treatments.

Economic Evaluation:

- Evaluate the economic feasibility and effectiveness of various control options to choose the most cost-effective strategies for nematode management.

Long-Range Planning:

- Utilize computer-assisted planning tools to develop long-term (3-5 years) nematode management strategies. Long-range planning can help optimize control measures, crop rotation schedules, and other interventions to sustainably manage nematodes over time (Ferris et al., 1993).

13.12.4 Management of Root-knot and Stem and Bulb Nematodes on Alfalfa

- Choose planting sites with good soil drainage, adequate sunlight, and proper soil pH to support healthy alfalfa growth and reduce nematode pressure.
- Start with certified seed that is free from nematode infestations.
- Maintain clean equipment during planting and cultivation to prevent the spread of nematodes between fields.
- Manage irrigation practices to avoid waterlogging and excess moisture in the soil,
- Implement effective weed management practices to eliminate weed hosts that can serve as alternative hosts for nematodes.

- Select alfalfa varieties that exhibit resistance or tolerance to nematodes

- Rotate alfalfa with non-host crops to disrupt the nematode life cycle and reduce nematode populations in the soil.

- Include fallow periods in the cropping system to help reduce nematode populations and break their life cycle.

- Consider chemical control options for managing nematodes when populations are high and other cultural practices are not sufficient (Westerdahl and Frate, 2007).

13.12.5 Preventing Peach Tree Short Life (PTSL) Influenced by Ring Nematode and *Pseudomonas syringae*

- Before planting, apply lime to adjust the soil pH in the top 20 cm to the optimal range of 6.0-6.5.

- Break up hardpan through subsoiling to improve water infiltration, drainage, root growth, nutrient uptake, and the diffusion of nematicides.

- In sandy soils with a history of peach trees or where root-knot nematodes are problematic, consider pre-plant soil fumigation

- Plant peach trees certified to be free of nematodes to reduce the risk of nematode infestations and other diseases.

- Opt for rootstocks like Lovell or Halford for planting, known for their susceptibility to root-knot nematodes, necessitating pre-plant fumigation in affected areas.

- Apply nutrients and lime as recommended based on soil tests, foliar analysis, and local guidelines to ensure proper nutrient levels for peach tree health and growth.

- Prune trees as late as possible, preferably after 1 February, to reduce the risk of PTSL, especially in orchards with a history of the disease. Cease summer pruning by 15 September to avoid tree stress.
- Use recommended herbicides for effective weed control. If mechanical cultivation is employed, ensure shallow cultivation to prevent root injury to peach trees.
- After pre-plant fumigation, monitor nematode populations annually and apply post-plant nematicides if ring nematode populations increase in the orchard.
- Promptly remove and destroy all dead or dying trees from the orchard to prevent the spread of diseases and pests (Nyczepir, 1989).

13.13 Summary

In many scenarios, fumigation and fungicides are still mainstays of management programs. However, as reviewed in this chapter, sustainable alternatives in the areas of improved diagnosis, prevention, physical, biological and cultural methods are seeing increased usage and integration into management programs. Notable among these are molecular diagnostics, improved understanding of biology and population dynamics, online databases and computer assisted modeling, newly developed resistant varieties, grafting of annual crops, trap cropping, biofumigation, new equipment for application of steam, remote sensing, commercially available biological controls and nematicides with new modes of action, solarization and anaerobic soil disinfestation.

	(a) EXTRACTION METHOD			
NEMATODE	ELUTRIATOR-SUGAR CENTRIFUGATION (NUMBER/LITER SOIL)	BAERMANN FUNNEL (NUMBER/LITER SOIL)	CHEESE CLOTH LINED FUNNEL (NUMBER/LITER SOIL)	ROOTS ON MIST (NUMBER/GRAM)
LESION	2,750	7,200	280	1.6
RING	1,000	20	150	0
DAGGER	150	260	350	0

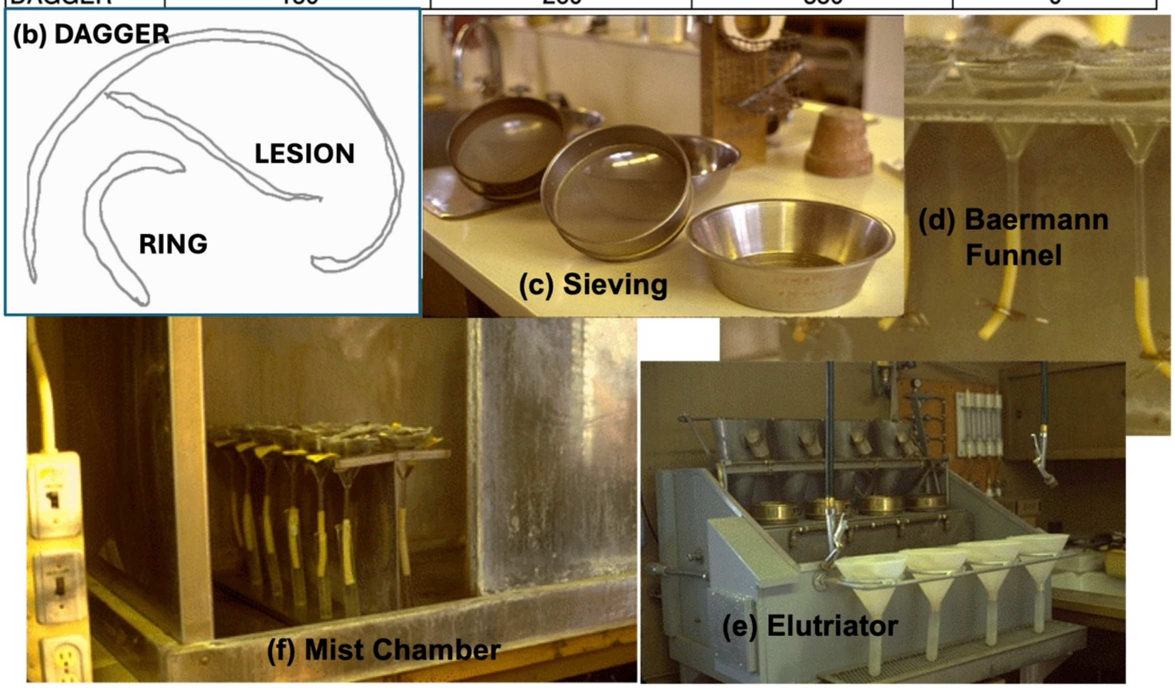

Figure 1.

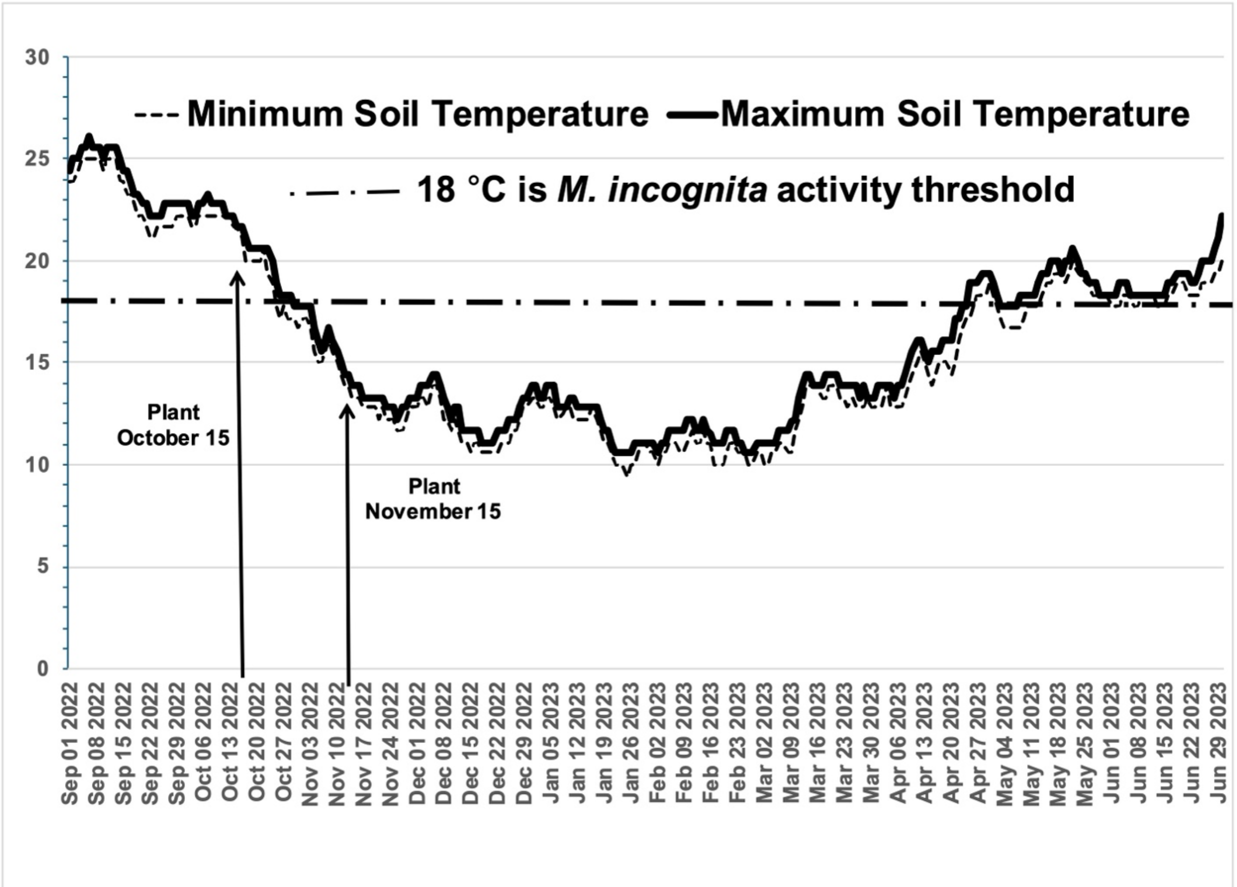
--- Minimum Soil Temperature —Maximum Soil Temperature
18 °C is M. incognita activity threshold
Plant
October 15
Plant
November 15
30
25
20
15
10
5
0
Sep 01 2022
Sep 08 2022
Sep 15 2022
Sep 22 2022
Sep 29 2022
Oct 06 2022
Oct 13 2022
Oct 20 2022
Oct 27 2022
Nov 03 2022
Nov 10 2022
Nov 17 2022
Nov 24 2022
Dec 01 2022
Dec 08 2022
Dec 15 2022
Dec 22 2022
Dec 29 2022
Jan 05 2023
Jan 12 2023
Jan 19 2023
Jan 26 2023
Feb 02 2023
Feb 09 2023
Feb 16 2023
Feb 23 2023
Mar 02 2023
Mar 09 2023
Mar 16 2023
Mar 23 2023
Mar 30 2023
Apr 06 2023
Apr 13 2023
Apr 20 2023
Apr 27 2023
May 04 2023
May 11 2023
May 18 2023
May 25 2023
Jun 01 2023
Jun 08 2023
Jun 15 2023
Jun 22 2023
Jun 29 2023

Figure 2.

Figure 3.

13.14 References

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