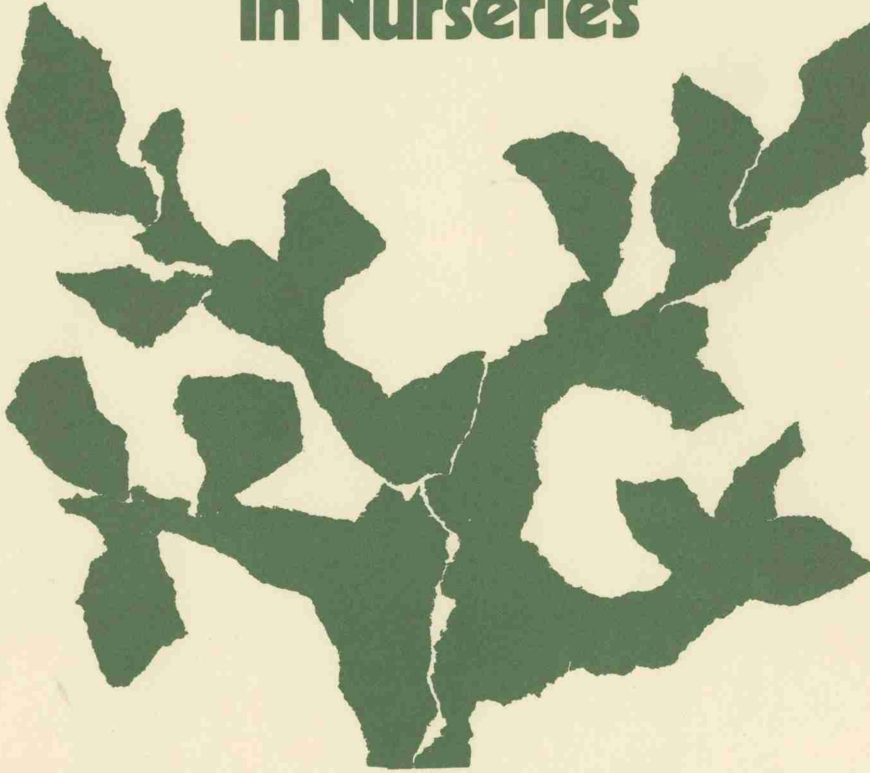
Diseases of Woody Ornamental Plants and Their Control in Nurseries



DISEASES OF WOODY ORNAMENTAL PLANTS AND THEIR CONTROL IN NURSERIES

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Introduction

R. K. Jones

Diseases are a constant threat to woody ornamental plants being grown in nurseries in the southeastern United States. A plant disease is the interaction of a pathogen, a susceptible host plant, and favorable weather conditions which results in an abnormal change that reduces the plant's value. On food and fiber crops, diseases usually reduce yield. With ornamental plants, diseases can cause loss by reducing quality as well as killing the entire plant.

Diseases reduce plant growth rate (stunting), uniformity, appearance, and plant quality. Diseases can also kill plants. This results in reduced operating efficiency because the cost of pesticides, pesticide application equipment and application labor are additional costs for the nurseryman. Plants may become diseased in the nursery and then be transplanted to the landscape where they may soon die. The sale of diseased plant material that dies soon after transplanting can damage the nursery's reputation and decrease future sales. A new plant pathogen could also be introduced into the planting site on nursery stock and could possibly be the first introduction of that pathogen into a state.

Many plant diseases result from a combination of factors, some living (biotic) and others non-living (abiotic). This publication emphasizes those diseases caused by biotic agents called pathogens (such as fungi, nematodes, bacteria, viruses and mycoplasm). The symptoms, effects of environmental conditions on development, spread and control will be discussed for many of the most important diseases affecting woody ornamentals in commercial nurseries. Diseases of woody ornamentals caused by these pathogens are contagious and, if not properly controlled, can result in severe losses. The information presented in this publication will help you identify and have a better understanding of diseases so that losses in your nursery can be minimized. A total disease control program for woody ornamental nurseries will be stressed. Chemicals for disease control and strategies for their use will be discussed. Specific chemical controls, however, are not recommended in this publication. These must be obtained locally.

Disease Development

Charles Hadden and R. K. Jones

Disease occurrence and severity in plants depends on several requirements. Foremost is the presence of a pathogen. The quantity and virulence (ability to cause disease) of this pathogen directly affects the severity of the disease. Since a disease is the result of an interaction between a pathogen and a host plant, it is necessary that a susceptible host plant must also be present. The number of susceptible host plants present and their level of susceptibility also limits the severity of a disease.

Disease development requires a certain set of environmental conditions. These environmental conditions include humidity, temperature and light. If they are suitable for disease development, they are termed a favorable environment. Some diseases can develop under a wide range of environmental conditions, whereas, others are more particular.

The severity or intensity of a disease will be greatest when a large quantity of a highly virulent pathogen occurs in a large number of highly susceptible host plants and a highly favorable environment occurs at the same time. The interrelationship between the pathogen, the susceptible host and a favorable environment is graphically expressed as a pyramid (Fig. 1). Disease development only occurs when a pathogen, susceptible host and a favorable environment occur simultaneously.

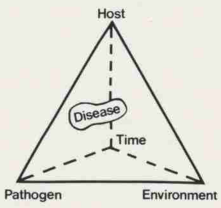

Figure 1. Disease pyramid illustrating the interrelationships between host, pathogen, environment, time and disease development.

Finally, time also influences disease development. The longer the susceptible host plant remains in the nursery, the greater the chance is for a favorable environment and the pathogen to be present at the same time and thus the disease develops. As soon as the first plant becomes infected, the pathogen begins to reproduce and spread to other susceptible plants within a block of susceptible host plants. During periods when environmental conditions favor disease development, disease incidence and severity increases as compound interest.

Under nursery production conditions in the Southeast, the proper combination of conditions necessary for disease development too often exists during much of the year. Large blocks of highly susceptible plants at several stages of growth are present at one location. Plants to be grown in a nursery are chosen more by what the grower can sell than what is resistant to disease. Plants are forced to grow rapidly and are often crowded. The weather is generally mild with adequate to excessive water due to rainfall or irrigation. The plants grown are perennials that stay in the production area several years. The most variable factor in the pyramid is the pathogen—its presence or absence.

All of the conditions necessary for development of some diseases occur only at certain times of the year. The conditions favorable for the development of fire blight of crab apple and leaf gall of azalea occur only for a short time in the spring. Other diseases, such as black spot of rose, can develop with a much wider range of conditions during the year and can be found on susceptible rose plants throughout the year in the South. Some diseases only affect certain parts of the plant or occur at certain stages of plant growth. Azalea petal blight only develops in the spring when azalea flowers are present; the pathogen is dormant during the rest of the year.

Infection by root disease pathogens is limited primarily by soil temperature and moisture. Soil conditions favorable for infection by several root pathogens of woody ornamentals occur throughout much or all of the growing season in southeastern nurseries. Once root infection takes place, the pathogen can continue to move in the root system under a much wider range of environmental conditions than is necessary for infection. This disease process may go on for several months before above-ground symptoms become obvious. Symptom expression on above-ground plant parts on plants with root diseases frequently occurs during or just after a stress period such as rapid growth, heat or seasonal change.

The seasonal plant disease development calendar (Fig. 2) is intended to help "predict" the occurrence of certain diseases. Only those diseases that usually occur at a particular time of year or at a particular stage of growth of the plant are listed. The line on the calendar indicates when the disease is likely to occur and when the pathogen may be active. The wider the line, the greater the probability of the disease occurring during that particular time period. The flowering dates are listed to help adapt the calendar to various locations in the region plus early or late seasons. Growth and disease development begins earlier in Florida than Virginia. Some of the diseases listed do not occur every year, do not occur across the region and can vary in severity from year to year. Some of the diseases that affect woody tissue may cause symptoms that remain after the calendar indicates the disease is no longer active; examples are fusiform rust on pine and fire blight on pear.

Figure 2.

Plant Disease Development Calendar

PLANTS	DEVELOPMENT OR DISEASE	PATHOGEN SCIENTIFIC NAME	PLANT PARTS AFFECTED	JAN.	FEB.	MARCH	APRIL	MAY	JUNE	JULY	AUG.	SEPT.	OCT.	NOV.	DEC.
Trees															
Crab Apple	Flowering														
	Cedar apple rust	Gymnosporangium sp.	leaves												
	Fire blight	Erwinia amylovora	shoots												
	Powdery mildew	Podosphaera leucotricha	shoots												
	Scab	Venturia inaequalis	fruit leaves												
Dogwood	Flowering														
	Anthracnose	Elsinoe corni	leaves flowers												
	Leaf spot	Septoria floridae	leaves												
	Scorch	Physiological	leaves												
Oak	Flowering														
	Anthracnose	Gnomonia veneta	leaves												
	Leaf blister	Taphrina caerulescens	leaves												
	Rust	Cronartium sp.	leaves												
Pine	Flowering														
	Eastern gall rust	Cronartium quercuum	branches trunk												
	Fusiform rust	Cronartium fusiforme	branches trunk												
	Needle cast	Hypoderma lethale	needles												
	Needle rust	Coleosporium sp.	needles												
Red Bud	Flowering														
Red Cedar	Cedar apple rust	Gymnosporangium sp.	branches												
Red Maple	Flowering														
	Anthracnose	Gloeosporium apocryptum	leaves												
Saucer Magnolia	Flowering														
Sycamore	Anthracnose	Gnomonia platani	shoots												
Woody Plants															
Azalea	Flowering														
	Leaf gall	Exobasidium vaccinii	leaves												
	Petal blight	Ovulinia azaleae	petals												
Camellia japonica	Flowering														
	Flower blight	Sclerotinia camelliae	flower												
	Leaf gall	Exobasidium camelliae	shoot												
Camellia sasanqua	Flowering														
	Leaf gall	Exobasidium camelliae	shoot												
Crape Myrtle	Flowering														
	Powdery mildew	Erysiphe lagerstroemiae	leaves flowers												
Forsythia	Flowering														
Pyracantha	Fire blight	Erwinia amylovora	shoots												
Rhododendron	Flowering														
	Leaf gall	Exobasidium vaccinii	shoots												
	Dieback	Phytophthora cactorum and other species	young shoots												
Rose	Flowering														
	Black spot	Diplocarpon rosae	leaves												
	Botrytis	Botrytis cinerea	flowers stems												
	Powdery mildew	Sphaerotheca pannosa	leaves												

Abiotic Causes of Plant Disorders in Nursery Crops

V. P. Bonaminio and R. K. Jones

In the production of both field grown and containerized ornamentals, numerous plant disorders develop which cannot be attributed to living or biotic organisms. Usually they are caused by non-living or abiotic entities. This can lead to considerable confusion since the symptoms of abiotic diseases often mimic those caused by pathogens. The following discussion is not meant to be all inclusive. Rather, it is meant to call attention to the more common causes of abiotic diseases of nursery crops.

Nutritional Disorders

There are 16 elements considered essential for the normal growth and development of higher plants which includes nursery crops. These are divided into the nine macro-nutrients (C, H, O, N, P, K, Ca, Mg, S) which are required in relatively large amounts and the seven micro-nutrients (Fe, Mn, Zn, B, Cu, Mo, Cl) which are required in relatively small or trace amounts. Nutrients must be supplied in the proper amount, ratio and form to insure normal plant growth and development. Nutrients applied in insufficient or excessive amounts result in mineral deficiencies or toxicities.

Consider the following nutritional disorders and how the symptoms compare to those resulting from biotic diseases. Compared to normal plants, plants grown with inadequate calcium are stunted, the terminal bud may be dead and leaves may have water-soaked blotches on them. The root systems are usually sparse, not well developed, brown in color and there are few, if any, healthy actively growing white feeder roots. Further, the tips of the roots are dead, black in color and often slimy.

On plants grown with insufficient nitrogen the young leaves are small and pale in color. Advanced symptoms include a uniform yellowing of foliage, beginning with the older leaves, followed by leaf drop.

Plant response to copper deficiency is a rosetting of leaves followed by necrosis of the terminal bud; the younger plant leaves may exhibit interveinal chlorosis but the leaf tips remain green.

The expression of a mineral deficiency or deficiency complex may also be a secondary plant response to a disease induced by biotic factors. Frequently root rot and nematode diseases restrict or completely inhibit a plant's ability to take up nutrients even when the nutrients are in abundant supply in the potting medium. Since diseased or dead roots cannot take up nutrients, mineral deficiency symptoms are usually first evident in the foliage.

The only reliable method for determining the cause and corrective measures for plant mineral deficiency symptoms is through a combination of root rot assays, nematode assay, foliar analysis and soil test.

Planting Media

A critical consideration in the production of nursery crops is the container medium or field soil in which the plants will be grown. The essential functions of a medium are to provide support for the plant and also to be a reservoir for water, nutrients and air. Under favorable conditions (proper balance of water, nutrients and air) root development is encouraged and proceeds at a rather rapid pace.

If plants are to be field grown, particular attention must be paid to site selection, soil type and texture. Avoid low areas where water drainage and soil aeration are notoriously poor. Such conditions inhibit root and top growth and render plants more susceptible to invasion by soil-borne pathogens, especially root rot organisms. Low areas also act as collecting basins for surface run-off water which can be contaminated with pathogens, weed seeds or chemicals. Soils in low areas are also usually cold, wet and high in organic matter, and therefore respond very differently to applications of fertilizers, fumigants and herbicides. Sandy loam, loam or clay loam soils on level or gently rolling terrain are ideal for field plantings. Always submit field soils for nutrient and nematode assay well in advance of planting so that corrective measures can be taken if needed. Soils must be relatively free of pathogens, weeds, weed seeds and soil insects. If they are not, then the soils should be fumigated or another planting site should be chosen.

In the production of containerized ornamentals in the Southeast, soil is seldom used as a potting medium. Cost, weight, availability, sanitation and variability are factors of concern. Most containerized ornamentals are being grown in soilless blended media. The components are bought in bulk and the medium is blended as needed by the grower. The major component is pine bark which may or may not be blended with shredded peat moss or sharp builders sand. With soilless media the choice of components and proportion of each is an individual decision. Since each of the individual components vary in physical, chemical and biological properties, the blended product is also variable. Further, depending upon the proportion of each component, the water holding capacity, drainage, aeration and pore space, cation exchange capacity, percentage organic matter and bulk density of the blend can be quite variable. For any particular grower, every attempt should be made to insure uniformity between batches of mix. A medium which is consistent in physical and chemical properties from one batch to the next will be less likely to present problems relative to fertility, watering, weed control and pathogen control. A potting medium not only affects root and plant growth but also the development of root pathogens. A good medium for ornamentals must favor the development of the crop plants over the pathogenic organisms. If soil is used in the medium or if there is a question as to contamination of any of the components, they should be fumigated prior to mixing. The extra effort at this time could well avoid disease problems once the crop plants are potted.

Media components (sand, bark, peat, perlite and vermiculite) are relatively free of pathogens when purchased, and once at the nursery they should be stored in an area where they will not be contaminated. A raised covered area is best but an outdoor concrete pad in the highest accessible part of the nursery is also suitable. They should not be stored downhill from the growing area or in low spots where contaminated surface run-off water collects. Neither should they be stored near the nursery refuse dump where dead plants, used media, old containers or innumerable other sources of contamination may be nearby. The same precautions must be taken with the blended medium. The media preparation, storage and potting areas should be the cleanest, driest and most sanitary in the nursery since all plants propagated or grown will come through this part of the nursery several times before being sold. It's much simpler and less costly to avoid disease problems through good sanitation than to attempt to control them after they develop. Two of the most successful control measures a nurseryman can follow are to exclude pathogens from the media and avoid conditions which favor their development.

pH

The pH of a potting medium is a measure of its' relative acidity (sourness) or alkalinity (sweetness). The scale used to measure pH goes from 0.00 (most acid) to 14.0 (most alkaline), with pH 7.0 being neutral. Soil pH critically affects microbial activity and nutrient availability. Soil microbes are most active between pH 5.5 to 10.0. Above or below this range their activity is severely reduced. Since the processes of nitrification and mineralization are dependent upon high microorganism activity, the availability of nitrogen from the soil for plant growth is also pH dependent.

Soil pH also affects the availability of nutrient elements. Even though applications of fertilizers are made to crop plants, the nutrient contained therein may not be available for uptake if the medium is not at the proper pH. As pH decreases below 5.0 the availability of nitrate nitrogen, phosphorus, potassium, sulfur, calcium, magnesium and molybdenum decreases. As pH increases above 7.0, the availability of phosphorus, iron, manganese, boron, copper and zinc becomes limited. It is essential therefore that the pH of the potting medium be monitored on a regular basis during the crop cycle and be maintained within a range favoring optimum nutrient availability. In many cases nutrient deficiencies, especially in nursery crops, are caused by improper soil pH. Thus, the unavailability of nutrient elements is the cause of the nutrient deficiency rather than the lack of fertilizers in the soil. Since pH adjustment is much less costly than applying more nutrients, it's just good business sense to have a soil analysis prior to fertilizing any crop, especially those which show deficiency symptoms. In most nurseries there is a tendency to let the potting medium get too acid rather than to alkaline.

Soluble Salts

The term soluble salts refers to the presence in a soil or potting medium of nutrient ions that are readily soluble in water. Calcium, chlorides, nitrates, phosphates, potassium and sulfates are but a few types of nutrient ions. Many of these ions are applied as plant nutrients in complete or trace element fertilizers. However, high concentrations of these salts can be damaging to plant roots.

There are two major causes of excess soluble salts in nursery crops. The first is heavy, infrequent and poorly distributed applications of fertilizers. Fertilizers should never be applied at more than the recommended rate and should be thoroughly distributed over the entire soil surface. Dropping fertilizer in piles beside field grown or containerized plants results in localized high soluble salts.

The second major cause is when insufficient water is applied during irrigation. For field grown plants, one-half to 1 inch of water should be applied at each irrigation. For containerized plants, enough water should be applied at each irrigation so that some moisture runs out of the drainage holes at the bottom of the containers. Other causes of high soluble salts are: 1) residues of applied fertilizer materials which are not used by plants in large quantities; 2) soil sterilization by chemicals or steam which releases salts previously tied up on soil colloids or by microorganisms; 3) high soluble salts in irrigation water; and 4) poor drainage.

Excess soluble salts damage plant roots through either burning or desiccation. Symptoms of soluble salt problems include wilting, chlorosis of foliage, stunting of root and top growth, and leaf burn or necrosis. High soluble salt problems can usually be corrected by leaching the soil or container medium with large volumes of water. Several applications a day apart may be necessary to reduce the salts to an acceptable level, especially if slow release fertilizers were the cause of the problem initially.

Watering and Water Quality

Proper watering (or irrigation) is one of the most critical routine tasks associated with nursery crop production. Irrigation cannot be done on a precise time schedule. In order to produce quality nursery crops they must be watered when they need it, and the demand is based upon many combined factors. During periods of high temperatures or winds when there is a high rate of water loss from plants and the medium in which they are growing, the plants will need frequent irrigation. Large plants and those with broad leaves usually require more frequent watering than small or narrow leaved plants. Containerized plants require more frequent irrigation than the same species planted in the field, even when they are the same size. Rooted cuttings, liners or small plants which are planted directly into 2 or 3 gal. containers will require less frequent watering than if they were potted into quart or gallon cans. Actively growing plants, especially those going through a flush of growth, require more water than dormant plants or those not in a growth spurt.

Overwatering, especially on heavy soils or poorly drained media, reduces aeration and results in plants "drowning." Visual symptoms on above-ground parts include a decrease in plant vigor and prolonged wilting of the foliage. In the early stages root systems become swollen or "fleshy" and brittle. Continued overwatering causes first the fibrous and later the larger roots to die. As they decline in vigor the roots progress in color from white to brown to black and become mushy or gelatinous to the touch. Overwatered container plants do not develop roots in the lower one-half to one-third of the media. Overwatering is frequently a problem in southern nurseries and this favors the development of root rot diseases.

When water is not applied frequently enough, even though an adequate amount is applied at any given watering, damage to the plants will ensue. Initial symptoms are a flagging or wilting of the foliage. Tender young shoots may also wilt. If conditions persist, newly expanding leaves may have necrotic margins and tips and vegetative and floral buds may abort. There is an overall loss of plant vigor. Plants subjected to repeated periods of water stress have poor root systems. There will be few if any feeder roots and if present they will be tough, stringy and brown in color. The development of a tap root system with few laterals will be very evident.

Improper watering, whatever the cause, has a marked and pronounced effect on plant roots. As roots decline in vigor and die, they become prime targets for entry by plant pathogenic organisms, especially root rotting fungi.

Environmental Stress

Environmental stress may be in the form of heat, cold, wind, water or light. Prolonged periods of unusually high or low temperatures can cause both foliar and root damage. Initial symptoms of heat stress include a loss of plant vigor and wilting of foliage and tender shoots. Unusually high temperatures which persist for prolonged periods of time may result in marginal necrosis of young or immature leaves. In severe instances there may be partial or complete leaf drop.

Cold injury or frost damage to plants initially causes the foliage to become flaccid and mushy with a water-soaked or greasy appearance. Shortly thereafter leaves may turn uniformly black in color and abscise. Plant stems are subject to splitting whenever previously frozen tissue is warmed rapidly. Conditions which favor frost splitting are extremely cold cloudy periods followed by either a rapid rise in atmospheric temperature or very bright sunny weather. When previously frozen plants are warmed rapidly, cell expansion is not as rapid as that of intercellular water. The result is that tremendous pressure builds up within the plant leading to an explosion within the tissue. The physical evidence of such events are the familiar frost cracks in plant stems.

Root systems are less susceptible to cold damage than are plant tops. In field plantings frost heave sometimes uproots young plantings in which case feeder roots may become completely detached from the main root system. Root injury from sub-freezing temperatures is more of a problem in containerized plantings than in field plantings, since the plants are being grown above ground. Periods of alternate freezing and thawing cause individual roots, especially those near the outer, windward and on the south side of containers, to burst. Affected roots take on a water-soaked appearance and upon thawing become gelatinous in texture.

High winds cause plant upheaval, limb and stem breakage and desiccation. Upheaval of entire plants results when high winds are preceeded by excessive rainfall or when plants fail to develop an adequate root system and become "top heavy." Wind is usually a secondary factor in limb and stem breakage. More often than not the affected limb or stem has been previously weakened by lightening or pathogens or has suffered from mechanical injury.

However, high winds can cause breakage of rapidly grown and naturally weak stems and limbs which have unusually dense foliage. Most wind-incurred plant damage is through desiccation. Whenever plants cannot take up sufficient water to replace that which is lost through evaporation and transpiration, desiccation occurs. Initial symptoms of desiccation are wilting of the younger leaves and tender shoots. If conditions persist there may be necrosis of the younger foliage followed by shriveling and leaf drop of the older foliage.

Plants respond to both light quality and quantity. In nursery operations plants are usually grown under natural light and quality or spectral distribution is seldom a concern. Problems do arise when plants previously grown under filtered or reduced light are suddenly subjected to bright light. Symptoms include a uniform "burning" of the foliage, especially the younger leaves and some leaf drop. Actively growing young shoots may wilt, newly expanding foliage may never fully develop to normal size and there may be some bud abortion. Plants grown in full light and suddenly subjected to reduced light may exhibit rapid and pronounced leaf drop beginning with the older foliage.

Snow, ice and hail should also be considered as abiotic causes of plant diseases. Excessive weight from accumulations of ice or snow can cause stems and branches to break and may even cause uprooting in shallow-rooted species, especially if the soil is moist or soggy. Hail damage is primarily to foliage in that it shreds or punctures leaves. On thin-barked species such as poplar, dogwood, pear and maple it may also cause vertical dagger-shaped wounds on stems and branches (Fig. 3).

Figure 3A. Hail damage to bark of 'Bradford' pear, 1 day after injury (R. K. Jones, NCSU).

Figure 3B. Hail damage to bark of 'Bradford' pear 6 months after injury (R. K. Jones, NCSU).

Air Pollution

Several gaseous atmospheric impurities are known to be phytotoxic to ornamental plants. In the Southeast there are fewer problems from air pollution damage to commercial nursery plantings than elsewhere in the country. However, nurserymen with production facilities in or around metropolitan or industrial centers or even at great distances from pollution sources, may occasionally see air pollution damage on their plants.

There are two classifications of air pollutant injury to plants: acute and chronic. Acute injury is less prevalent, and occurs when plants are subjected to an extremely high level of pollutant for a short period of time thereby producing visible foliar symptoms. Chronic injury is of more concern and occurs when ornamentals are subjected to low levels of pollutant for long periods of time. Chronic injury may not become evident for several years but it results in a decreased growth rate, and usually predisposes plants to damage by environmental or pathogenic stress. Phytotoxic air pollutants that concern nurserymen include ozone, sulfur dioxide, fluorides, ethylene, chlorine, peroxyaetcyl nitrate (PAN), hydrogen chloride and nitrogen oxides.

Chlorine, ethylene, fluorides, hydrogen chloride and sulfur dioxide are usually considered point source pollutants in that injury from them usually occurs within 10 miles from where they are released into the atmosphere. Damage from ozone and PAN can occur for many miles from their source depending upon such environmental factors as wind direction, velocity and humidity.

Ozone and PAN are formed in the atmosphere from chemicals released primarily from automobile exhausts. The burning of some fossil fuels may also result in their formation and damage to plants usually occurs in and around large metropolitan areas. On conifers, needle tip chlorosis, necrosis and chlorotic banding and mottling are symptoms of ozone injury. On broadleaves ozone injury is characterized by a chlorotic flecking or stippling of the upper leaf surface. PAN injury produces a wet looking or silvery color on the underside of leaves; the tips of young or actively expanding leaves may be white in color or necrotic. Ozone is the most damaging air pollutant in the Southeast.

Sulfur dioxide induces tip necrosis and banding on conifer needles and large areas of interveinal chlorosis on the upper and lower surfaces of broadleaves. The major source of sulfur dioxide is from the combustion of fossil fuels, particularly coal high in sulfur.

Fluoride is a natural component of many minerals. It may be released into the atmosphere when those materials are heated to high temperatures such as in the manufacture of bricks, ceramics, glass and phosphate fertilizers. It may also be produced in steel mills and in the reduction of aluminum ore. Exposure to fluorides produces leaf tip and marginal necrosis in broadleaves and conifers.

Damage to plants from chlorine is usually caused by accidental leakage from storage tanks, industrial production facilities and water purification plants. It is also released from the commercial production of textiles (especially cotton fabrics), flour and glass and from the burning of plastics. Symptoms of chlorine damage include complete leaf chlorosis.

Ethylene is a naturally occurring compound in plants and is given off in large quantities from ripening fruits and during foliar decay. It is also a by-product of combustion of fossil fuels. In heated facilities where plants are being produced ethylene damage can be caused by incomplete combustion of wood or fossil fuels and by poor ventilation. Ethylene damage may also be induced when plants are overwintered or otherwise placed in facilities in which fruits have been or are currently being stored. Exposure to ethylene causes premature plant senescence and leaf drop.

Chemical Injury

Most injury from chemicals to nursery crops is the result of misuse or misapplication of the materials. Often problems can be avoided if time is taken to read, understand and follow label directions. Materials which are commonly misused and cause plant damage include herbicides, wood preservatives, paints, solvents, insecticides, growth regulators, fungicides and fumigants. The materials themselves are seldom the primary culprits—rather it is the person applying them.

The specific symptoms of chemical injury are as varied as the number of chemicals available. General symptoms include general plant decline, foliar burn, leaf necrosis or burning of the root system. To determine the cause of induced chemical injury and to define the pathogen, it is necessary to know the complete record of all materials used during the life of the crop. Chemical injury to plants is probably the most difficult to define and can often be confused with that caused by pathogenic organisms or infectious diseases.

Mechanical Injury

Mechanical injury is classified as physical damage to any part of a plant. It can be incurred by biological entities such as mice, rabbits and other rodents which feed on young shoots, leaves, bark and roots of nursery crops. Damage from these sources can occur during the growing season, but most often occurs during the fall and winter months when other food sources are unavailable. Evidence of rodent damage includes missing plant parts such as roots, stem ends, leaves and bark.

Another source of mechanical injury is from people and equipment during normal, but carelessly conducted, cultural practices. Cultivating too close to or too deeply around field plantings with power machinery can destroy root systems and place plants in a state of decline. Careless use of hand hoes usually results in injury to bark or plant stems at or near the soil line. Occasionally complete girdling of the stem may occur. Power mowers, especially in the hands of irresponsible personnel, can be lethal to nursery crops. Small plants are easily mowed off (especially in weedy nurseries). Plants in general are skinned and debarked and flying debris can puncture foliage as well as bark. In some situations mechanically induced damage can be easily mistaken for biotically induced leaf spots, stem dieback, canker and root rots.

Species Adaptability

Not all plants perform well in all situations and this can sometimes be an abiotic cause of disorder. In propagation beds, junipers and most narrow-leaved evergreens require less misting than broad-leaved species. Continuous overmisting and a slow drainage media can result in rotting of most species. Plants exported out of their climatic zone and into colder or warmer zones never perform satisfactorily. Not all species are adapted to dry, wet or seaside conditions. Rhododendrons and azaleas are almost guaranteed to die if planted in soils which are compacted or remain soggy for long periods of time. Shallow-rooted species such as the Japanese hollies cannot tolerate droughty or wet soils. When producing plants or utilizing them in a landscape, it must be remembered that there are suitable plant materials for almost all situations but that not all plants are suitable to be grown in all situations.

Biotic Causes of Woody Ornamental Diseases

The general characteristics of biotic causes (living pathogens) of diseases of woody ornamental plants, such as, fungi, bacteria, nematodes, viruses and mollicutes are discussed in this section (Fig. 4).

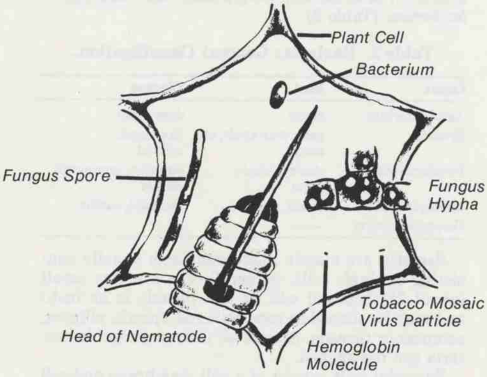

Figure 4. Illustration showing relative size of various types of plant pathogens in relation to a plant cell (NCSU).

Fungi

Wirt H. Wills

Fungi are living organisms classified by some taxonomists as plants, by others in a separate kingdom exclusive of both plants and animals. They lack chlorophyll and hence cannot synthesize their energy from CO_2 and water as do the green plants. They are, however, capable of a wide range of synthetic and degradative activities once provided with acceptable energy sources such as simple or complex carbohydrates. It is in the performance of these metabolic activities that fungi cause diseases in plants.

Some fungi are strictly saprophytic in their nutrition; they gain all their nutrients from dead organic debris in their environment. Other fungi are able to gain their nutrients in nature only from a living host; these are called obligate parasites. A third group, which contains most plant pathogenic forms, are able to live in both worlds. They colonize living plant tissues at times, and under other circumstances, they live a saprophytic existence.

The mycorrhizae form a special case of association between fungi and green plants. The term literally means fungus root, and describes an association in which the fungus grows in and on the higher plant roots and may assist the plant in uptake of nutrients, especially phosphorus. Some of these associations have also been described as harmful to the plant.

It is estimated that there are more than 100,000 species of fungi, of which about one-third have been described. Perhaps 10 to 20 thousand of them are plant pathogens. Since all other species of plant pathogens such as bacteria, nematodes and virus together may number less than 1 thousand, the fungi assume a predominance among plant pathogens.

Fungi are recognized by the structures they form. The vegetative or assimilative body of a fungus is usually a *thallus* composed of strands and masses of microscopic threads, collectively called the *mycelium*. Hard compact aggregations of fungal mycelium are known as *sclerotia*. These are usually survival structures.

If compact masses of fungal tissue contain reproductive structures in cavities, they are called *stroma*. Typically, a fungus reproduces by the formation of *spores* of various types. Spores may be formed asexually in which no nuclear fusion occurs (*conidia* and *sporangiospores*) or by a sexual process involving nuclear fusion (*oospores*, *zygospores*, *ascospores* and *basidiospores*).

The presence or absence of sexual spores and the type of sexual spores produced provide the means of classifying the fungi. An understanding of the classification of fungi can help growers diagnose and control diseases. Diseases caused by similarly classified fungi (Table 1) often produce similar symptoms, have similar mechanisms of spread and infection and are controlled by similar methods and fungicides. Also, plant family groupings can provide clues about disease susceptibility.

Plant pathogenic fungi infect and colonize all parts of green plants; roots, stems, leaves, flowers, seeds and fruits. Some are highly specialized, occurring only on the roots, the conducting tissues or on flowers. Others are omnivorous, attacking all tissues. The dead remains of diseased plants are often reservoirs of *inoculum* from which the fungus may later grow and attack susceptible plants. Some plant pathogenic fungi survive for long periods of time in the soil in organic debris or as spores or sclerotia in the soil.

Under proper conditions fungi in the soil or on old plants, dead or alive, may produce spores. These spores constitute the *primary inoculum* which can be carried by wind, water, insects or man to healthy plants. When these plants become *infected* (the fungus gains entry) and *colonized* (the fungus becomes established in the host plant) the fungus usually forms reproductive structures with spores (*secondary inoculum*). These serve to infect more plants and produce plant disease epidemics. Later in the season the fungus may become dormant and overwinter before producing primary inoculum the next spring. There are, of course, many variations on this theme.

Table 1. Fungi: General Classification by Groups.

Genus	Important Host	Disease
I. Phycomycetes—water molds and downy mildews, sexual stage oospore.		
Pythium	many	damping-off
"	"	root rot
Phytophthora	azalea, rhododendron, conifers, many others	root rot
	rhododendron, pieris	dieback
Peronospora	rose	downy mildew
II. Ascomycetes—sexual stage ascospore.		
Sphaceloma	rose	powdery mildew
Glomerella	camellia	canker
Ovulinia	azalea	petal blight
III. Basidiomycetes—rusts and smuts, sexual stage basidiospore.		
Exobasidium	azalea, camellia	leaf gall
Pucciniastrum	azalea, hemlock	leaf rust
Gymnosporangium	crab apple, juniper	cedar-apple rust
IV. Deuteromycetes—no sexual stage found, most plant pathogens occur in this group but probably have an Ascomycete sexual stage.		
Odium	many	powdery mildew
Colletotrichum	many	anthracnose
Phomopsis	azalea	dieback
V. Mycelia sterilia—mycellial stage of Basidiomycete lacking asexual spores.		
Rhizoctonia	many	damping-off, web blight
Sclerotium	aucuba	southern stem blight

Bacteria

John R. Hartman

Bacterial diseases such as crown gall and fire blight are familiar to many growers of woody ornamental plants. These diseases, as well as leaf spots and cankers caused by bacteria, can cause severe losses of plant materials in nurseries and landscapes. Most bacterial diseases of plants are caused by one of five genera of bacteria: *Erwinia*, *Pseudomonas*, *Xanthomonas*, *Corynebacterium* and *Agrobacterium* (Table 2).

Table 2. Bacteria: General Classification.

Genus	Host	Disease
Agrobacterium	many	crown gall
Erwinia	pear, crab apple, etc. many	fire blight, soft rot
Pseudomonas	cherry, peach, plum	leaf spot, gummosis, canker
Xanthomonas	peach, plum	leaf spot, canker
Corynebacterium	——	——

Bacteria are simple microorganisms usually consisting of single cells. Bacterial cells are very small (about 25 thousand will fit side-by-side in an inch) and may be shaped as rods, spheres, spirals, ellipses, commas or threads. Almost all plant pathogenic bacteria are rod-shaped.

Bacterial cells consist of a cell membrane and cell wall which enclose a cytoplasm. In this cytoplasm is the complex mixture of proteins, lipids, carbohydrates, nuclear material, and many other organic compounds, minerals and water which are essential for life processes. Most bacteria produce a layer of slime which adheres loosely to the outer surface of the cell wall. Bacteria reproduce by dividing (fission), growing and dividing again, a process that can occur as quickly as every 30 to 60 minutes resulting in large numbers of bacteria in a short time (Fig. 5).

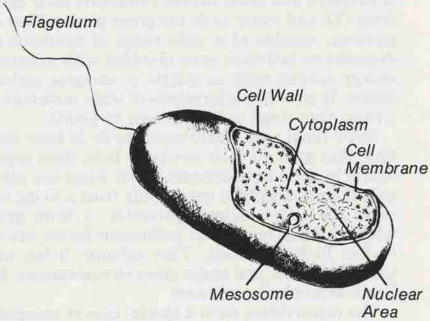

Figure 5. Illustration of plant parasitic bacterium (Pi-Yu Huang, NCSU).

Most plant pathogenic bacteria develop partly in the host plant as parasites and partly in the soil or dead plant material as saprophytes. The movement of plant pathogenic bacteria from soil or from diseased to healthy plants is carried out mainly in or on water (rain splash, primarily), insects, other animals and man. Bacteria that cause plant disease may be moved long distances on or in infected seed and plant materials. Infection of a plant by bacteria usually occurs when bacteria invade the host through wounds or natural openings such as leaf stomates. Once inside the plant, the bacteria may produce enzymes and toxins which cause the infected plant tissue to collapse and die. Other plant pathogenic bacteria invade the host vascular system which causes the plant to wilt. Others induce the plant to form tumors.

Nematodes

N. A. Lapp

Nematodes are microscopic worms that feed on man, his animals and his plants. It is generally believed that nematodes feed on most if not all plants whether they be cultivated plants, weeds or naturally occurring plants growing in uncultivated areas. Many plants, especially those occurring naturally in uncultivated areas, have adapted to this feeding such that they grow and reproduce even in the presence of nematodes. Most of the plants that man tries to cultivate, however, are not able to grow and reproduce well in the presence of large numbers of certain nematodes. Man has tended to select plants for the quality of their product rather than for their ability to withstand the attack of plant parasitic nematodes (Table 3).

Table 3. Nematodes: Genus and Common Name of Plant Parasitic Nematodes and Examples of Woody Ornamental Plants Attacked.

Genus	Common Name	Host
Criconemella	ring	many
Meloidogyne	root knot	Japanese holly, many others
Pratylenchus	lesion, meadow	boxwood, juniper
Trichodorus	stubby root	Taxus
Tylenchorhynchus	stunt	azalea
Xiphinema	dagger	many

Nematodes are typically worm shaped (Fig. 6) and are composed primarily of a digestive system and a reproductive system. All plant parasitic nematodes have a hollow spear (or stylet) located at the anterior end of the body which they insert into the plant to feed. The median bulb of the esophagus is a pumping organ which aids in sucking food from the plant. The food passes into the intestine where most of the digestion takes place. Excretion of waste products is through the anus near the posterior end of the body.

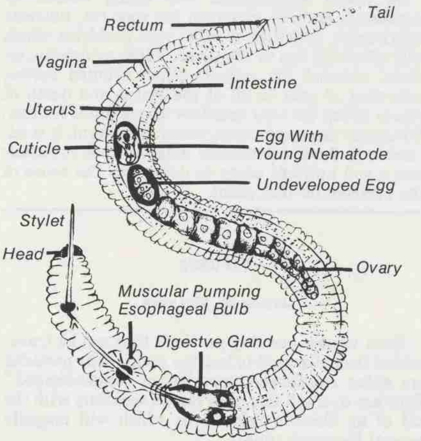

Figure 6. Illustration of "typical" plant parasitic nematode (NCSU).

The reproductive system of the female is composed of one or two ovaries, oviducts and a vulva which usually opens near the middle of the nematode. The male reproductive system is composed of testis, the associated ducts and some clasping organs (spicules and bursa) which aid in holding the female during the transfer of sperm. Although many nematode species have both males and females and reproduce sexually, some of the most serious parasites have only females and produce young without the use of male sperm.

Although most nematodes are worm shaped, there are some species that have different shapes. Some nematodes, such as the root knot and cyst nematodes, have a fat, rounded shape whereas others such as the ring nematode have a very ornamental body covering. Others such as the sting nematode are very long.

Nematodes cause damage to plants in a variety of ways. Some feed at or near the root tip causing the root to stop growing. Others feed on root cells causing death of these cells, resulting in a distorted, malformed root system. Nematodes that feed on the outside of the root are called ectoparasites. The endoparasites may either move through the root, feeding on cells as they move, or they may establish a feeding site in one location and remain there.

The root lesion nematode that attacks boxwoods as well as other woody ornamentals is an example of a nematode that moves into the root and feeds at various locations in the root. The root knot nematode which attacks a wide variety of woody ornamental plants moves into the root and establishes a feeding site in one location. It remains at this location the rest of its life and causes the root to form a gall around the nematode.

On woody ornamentals root injury caused by nematodes is often mistaken for root rot, nutrient deficiencies, toxicities or other root problems which will affect the top of the plant. These symptoms include: reduced top growth, small stunted leaves, yellowing of part or all of the leaves and death of plants which are very sensitive to nematode feeding. Whenever these symptoms become apparent, it is advisable to have a nematode assay, a root rot assay and a soil nutrient assay to determine the cause of the problem on that plant.

Viruses

James M. McGuire

Plant viruses are tiny particles that can be transmitted from diseased to healthy plants. The particles are either rod-shaped, spherical or "bullet-shaped." They are so small that they can be seen only with the aid of an electron microscope which will magnify several thousand times.

Virus particles are composed of an inner core of nucleic acid, either ribonucleic acid (RNA) in most plant viruses or desoxyribonucleic acid (DNA), and covered with a protein coat. The nucleic acid is the part which infects plant cells and directs the cell to manufacture virus particles rather than normal cell materials. The protein coat protects the nucleic acid from inactivation and may function to attach the virus to a vector or to the plant cell during transmission.

The replication of viruses in the plant results in symptoms such as mosaic and/or ring spot patterns of green and yellow in leaves, leaf distortions, dead rings or spots on leaves, flower variegation and/or distortion, stunting, rosetting and sometimes decline in vigor of plants.

Viruses are transmitted and perpetuated by vegetative propagation. The virus is usually systemic throughout the plant. Therefore, all plants that are propagated by cuttings or grafted will be infected if propagation wood came from a diseased plant.

Most viruses also have a vector organism that acquires the virus during probing or feeding on an infected plant and transmits it during subsequent feeding on a healthy plant. These vectors may be insects such as aphids, leafhoppers and thrips, nematodes or fungi. Each virus has a specific type of vector. In woody ornamentals, aphids and nematodes may be the most important vectors.

Virus host combinations are very specific. Some viruses infect many different hosts, but most viruses infect only a few specific plants (Table 4). Virus diseases are common and damaging in members of the rosaceae (Rose family) and particularly those that are grafted.

Table 4. Viruses: Common Names of Some Viruses Important in Woody Ornamental Nursery Plants.

Virus Name	Vector	Some Hosts
Tobacco ring spot virus (TRSV)	nematodes, grafting	Nandina Dogwood
Tomato ring spot virus (TmRSV)	nematodes, grafting	Dogwood Peach Cherry Hydrangea
Rose mosaic virus (RMV)	grafting	Rose
Rose spring dwarf virus (RSDV)	grafting	Rose
Camellia yellow mottle virus (CYMV)	grafting	Camellia
Cucumber mosaic (CMV)	aphids, grafting	Nandina Dogwood
Prunus necrotic ring spot virus (PNRV)	grafting	Cherry Apricot Peach Almond Rose

Mollicutes

James M. McGuire

Mycoplasmas and spiroplasmas, which make up the mollicutes, are primitive, membrane-bound organisms that lack a cell wall. Like bacteria, they have cytoplasm and genetic material but not in an organized nucleus. They are between viruses and bacteria in size and are often submicroscopic, which means they can be seen only with the aid of an electron microscope. They reproduce by simple cell division.

Mollicutes have been associated with a large number of diseases, formerly thought to be caused by viruses, in many plant species. The main symptoms of these diseases are gradual, uniform yellowing or reddening of leaves, distortion and decrease in leaf size, stunting, deformity, greening of flowers and buds, excessive number of shoots forming witches' brooms and sometimes decline and death of plants. Diseases in a number of woody plants and in herbaceous ornamentals are caused by mollicutes, but little is known of their importance as disease causing agents in woody ornamentals (Table 5).

Mollicutes invade phloem cells (food conducting tissue) in diseased plants. Most mollicutes are transmitted by leafhoppers which feed on host phloem. They can also multiply inside the insect's body, which means that when a leafhopper acquires a mollicute it will always carry it and will pass it through eggs to offspring. Mollicutes are also perpetuated by vegetative plant propagation and grafting.

Table 5. Mollicutes: Some Important Diseases in the Woody Ornamental Nursery Industry.

Type Pathogen	Disease Name	Method of Transmission	Hosts
Mycoplasma	Lethal Yellowing	Leafhoppers	Palms in several genera: *Caryota, Chrysalidocarpus, Cocos, Phoenix, Veitchia*, others
Rickettsia	Phoney Peach	Leafhoppers Grafting	Peach
Rickettsia	Pierce's Disease	Infected stock plant, Sharpshooter Leafhoppers	Grape

General Diseases

Diseases that occur on many different hosts are discussed in this section. The symptoms of the disease will be covered under the section on specific hosts.

Phytophthora Root Rot— *Phytophthora cinnamomi*

D. M. Benson

Several species of Phytophthora cause root rot in ornamentals, including *P. cinnamomi, P. citricola, P. citrophthora, P. drechsleri, P. lateralis* and *P. nicotianae* var. *parasitica*. However, *P. cinnamomi* will be used as a type-species to illustrate Phytophthora root rot. *Pythium* is another genus of water mold fungi that cause root rot of woody ornamentals. *Pythium* spp. tend to be more damaging on young plants, rooted cuttings or liners rather than larger woody plants. There are several species of *Pythium* with a very wide host range.

The fungus *Phytophthora cinnamomi* was first described by Rands in 1922 as a disease on cinnamon in Sumatra. In the 58 years since that time, *P. cinnamomi* has been reported on over 900 hosts worldwide. Crandall and Gravatt speculated that *P. cinnamomi* had its origins in Asia since no hosts have been reported from China, Korea, Japan, Vietnam, Laos, Cambodia or Thailand. However, nearby Indonesia has a wide distribution of *P. cinnamomi* from which early explorers and traders in the 15th and 16th centuries may have distributed the fungus on plants and in soil in Africa and subsequently to Europe and the Americas. Eventually, the fungus also reached India, Australia, New Zealand and the Pacific Islands.

Introduction of *P. cinnamomi* to North America probably occurred through the ports of Savannah and Mobile sometime prior to 1780. The fungus spread through the South by attacking roots of native chestnut, chinquapin and ericaceous plants such as rhododendron, azalea, leucothoe and blueberry.

Normally the fungus attacks hosts in low, wet, poorly drained areas first and then gradually moves to plants on higher and drier sites. Today, many important nursery crops are attacked by *P. cinnamomi*: andromeda (*Pieris*), arborvitae, aucuba, azalea, *Camellia, Chamaecyparis, Cunninghamia, Daphne*, deodar cedar, dogwood, Forsythia, Fraser fir, hemlock, Japanese holly, juniper, *Pittosporum, Podocarpus, Rhododendron, Stewartia*, white pine, yew and others. Phytophthora root rot is the most important disease in nursery production of woody ornamentals in the Southeast and the entire United States.

Symptoms

Severity of Phytophthora root rot and, hence, expression of symptoms associated with the disease varies among hosts. Hosts in which slight root rot develops may exhibit only mild symptoms such as slight stunting of the foliage and necrosis (death) of feeder roots. In this case, the disease may go unnoticed by the grower.

When root rot is severe, symptoms become more noticeable. Symptoms on the foliage include chlorosis (yellowing), marked stunting, wilting and death. Temporary disappearance of interveinal chlorosis may occur if foliar applications of iron are made—even when adequate iron is available in the soil as infected roots cannot adequately supply iron to the foliage. Plants may be stunted in overall size and individual leaves may be dwarfed. This is particularly a characteristic symptom on azaleas, white pine and shortleaf pine. Wilting occurs just prior to plant death and results in leaves that droop on the stems, even when water is applied to the plant. Cutting through the bark on the main stem of a wilted plant at the soil line often will expose a reddish-brown discoloration of the wood resulting from the advanced stage of root rot. Examination for discolored wood is recommended only on plants that can be sacrificed as considerable damage will be done to healthy plants by the knife cut.

Below-ground symptoms are also evident on plants with severe root rot. Examination of roots for rot is done by washing off the potting medium in a bucket of water. When root systems on diseased plants are compared with those from healthy plants, there is a greatly reduced root system on diseased plants. The color of roots of diseased plants will be reddish-brown to dark brown (Color Plate I, 1 and 8; Color Plate III, 5). Feeder roots may be completely lacking and coarse roots and the lower stem may be discolored. On plants with fleshy roots like aucuba and camellia, diseased roots may appear brown and water-soaked. Roots of healthy plants appear white, at least until secondary growth takes place in subsequent years, but the root tips still remain white.

Identification of *P. cinnamomi* as well as other *Phytophthora* spp. is based on cultural characteristics of the fungus, including growth habit and presence or absence of various spore-types that are observed both macroscopically and microscopically. Cultures of the fungus are obtained by placing small segments of infected roots on selective media in petri dishes. In culture, *P. cinnamomi* forms hyphae with swellings that can develop into chlamydospores in 5 to 7 days at 20° to 25°C. Sporangia develop in nature from mycelium, chlamydospores or oospores presumably stimulated by bacteria in the soil. Sporangia germinate to form zoospores that are motile in water films found in pores between soil particles (Fig. 7).

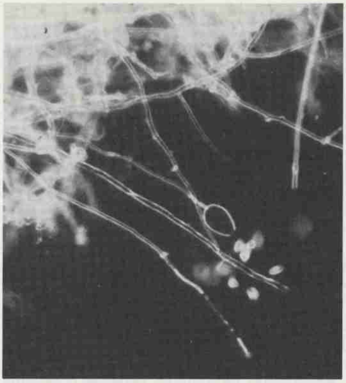

Figure 7. Microscopic view of sporangia of *Phytophthora cinnamomi* releasing zoospores. Zoospores swim to and penetrate the host root (D. M. Benson, NCSU).

Zoospores infect feeder roots of the host root system, attacking the root just behind the root cap. Infection of host roots occurs when certain environmental conditions are met. Soil temperature near 22°C is optimal but infection probably takes place from 15° to 28°C. Soil water is the critical factor in infection. Sporangia are most readily produced when the soil is just below saturation.

Phytophthora root rot is described as a "wet foot" disease of plants because of the requirement of wet soils for sporangium formation. Likewise, motile zoospores from sporangia are produced most abundantly when the soil is at saturation. Sporangia form in 4 to 8 hours under optimum conditions and zoospores can be released in 20 to 60 minutes. Any time that temperature and moisture conditions are favorable, zoospores can infect roots of susceptible plants. Generally, this period is March through October in North Carolina but will vary in other states (Fig. 8).

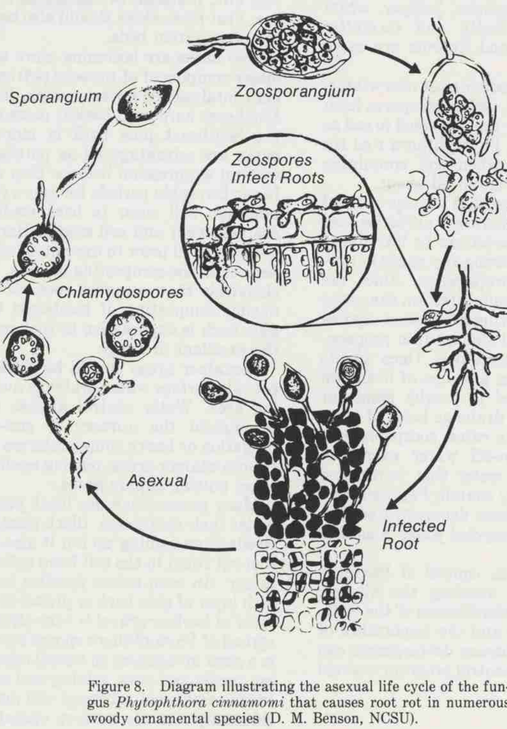

Figure 8. Diagram illustrating the asexual life cycle of the fungus *Phytophthora cinnamomi* that causes root rot in numerous woody ornamental species (D. M. Benson, NCSU).

Low soil pH reduces sporangium formation and hence the production of zoospores for infection. However, at pH values that inhibit sporangium formation, (3.5 to 4.0), plant growth is poor. Over the pH range optimal for growth of ericaceous hosts, (5.0 to 6.0), pH is not a factor in Phytophthora root rot.

Once infection occurs, rate of root rot development varies among hosts, depending on the size of the host as well as its relative susceptibility. Since the pathogen enters the feeder roots first and then spreads into the larger roots, the size of the root system and the total number of infection sites determine how fast the disease develops. For instance, rooted cuttings of azaleas were killed in 30 days, whereas 1-year-old plants in 1 gal. containers were killed in 5 to 7 months. Landscape-size plants may decline over a period of several years before they are killed by Phytophthora root rot.

Hosts vary in their susceptibility to Phytophthora root rot. In general, rhododendrons are more susceptible to root rot than azaleas and Fraser fir are more susceptible than white pine. Relative susceptibility of the more commonly grown ornamentals in order of decreasing susceptibility are Fraser fir, yew, rhododendron, azalea, *Camellia japonica*, juniper, white pine, dogwood, Japanese holly and *Camellia sasanqua*. Certain cultivars and hybrids are more susceptible than others.

Chlamydospores are the predominant overwintering structure of *P. cinnamomi*. Chlamydospores form in infected host roots and later are dispersed to soil as the infected root decomposes. Phytophthora root rot can spread in the nursery when soil containing chlamydospores or zoospores is moved about.

In the Southeast, *P. cinnamomi* is widespread and probably occurs in soils wherever nurseries are situated. Spores of *P. cinnamomi* can be introduced to susceptible nursery stock during any phase of nursery production, including propagation. Since the pathogen overwinters in the soil, infection can occur any time soil is introduced during production. Spores can be splashed from the soil surface into propagation beds or container-grown stock. Once plants become infected, they serve as a source of inoculum (spores) that can be splashed to nearby plants or move in water films through drainage holes of adjacent containers. During heavy rains, zoospores may be carried passively in run-off water over considerable distances. Run-off water that is recycled through the nursery pond may contain *Phytophthora* spores. However, it has not been demonstrated that disease is any greater when recycled water is used in irrigation.

As with most root diseases, control of Phytophthora root rot depends on avoiding the disease. Growers who appreciate the significance of the widespread nature of the fungus and the importance of drainage and soil water in disease development can design the necessary disease control program to avoid the disease.

Control

Control measures are based primarily on cultural practices and sanitation but the use of fungicides and resistant cultivars may be helpful. Cultural practices are by far the most effective measures for avoiding Phytophthora root rot. In the propagation house, direct contact of potting medium with the ground should be avoided by using screen-bottom benches. Taking cuttings high on the stock plants where soil has not splashed will avoid introducing the pathogen to the rooting bed. A rooting medium should be well drained and free of *P. cinnamomi*.

Prior to sticking cuttings and filling benches, all wood surfaces should be scrubbed down with a disinfectant. Containers used in potting operations should be new or fumigated prior to reuse. Avoid storing new containers directly on the ground.

A well-drained potting mix is absolutely essential for growing Phytophthora-susceptible ornamentals. Generally, soil should be avoided because it retains water too long, may contain spores of *Phytophthora*, and makes containers too heavy to handle conveniently. When soil is used as a component of the potting mix, it should be fumigated or steamed prior to use. Peat moss alone should also be avoided except in the propagation beds.

Tree barks are becoming more widely used as the major component of nursery potting media for growing containerized nursery stock. In the Midwest and Northeast hardwood bark is more available, while in the Southeast pine bark is more abundant. Tree barks are advantageous as potting components for root-rot suppression because they drain well. Hence, fewer favorable periods for spore formation and germination will occur in bark media than in slower draining peat and soil media. Hardwood bark must be composted prior to use but an additional benefit is derived in the composting process. Certain inhibitory chemicals that suppress root rot fungi are formed during composting of hardwood bark. Apparently, pine bark is suppressive to root rot fungi because of the excellent drainage.

Container areas should be graded prior to use to provide surface water drainage away from the growing area. Water control ditches should be located throughout the nursery so run-off water during irrigation or heavy thunderstorms is channeled away from container areas, potting media storage piles and other potting supply areas.

Many nurserymen use black plastic on the ground under their containers. Black plastic not only inhibits weeds from coming up but it also prevents spores of root rot fungi in the soil from splashing into the container. An even better practice is to apply a 3 to 4-inch layer of pine bark or gravel over the plastic. This layer of bark or gravel is very effective in preventing spread of Phytophthora spores between containers. It is a good investment to install concrete pads for storing media and pots, mixing and canning operations. In this way, root rot fungi will not be introduced accidentally into the nursery operation.

Plants should be grouped in the nursery by container size and water requirements. Many nurserymen fail to realize that proper irrigation is one of the most critical steps in nursery production to avoid root rot problems. Grouping plants by their water requirements avoids the danger of overwatering root rot susceptible plants. Commercially-available irrometers can be used to monitor soil-water status so that water is not applied until needed.

Present-day fungicides are used in disease control to prevent root infection. Granular fungicides for root rot control can be mixed with potting medium at canning or liquid fungicides are applied as drenches to the surface. In both methods of application the fungicide is effective only if: 1) it is applied prior to root infection; and 2) concentration of the fungicide remains at or above the effective rate. This latter requirement means that repeated applications will be needed, usually at 2 to 8 week intervals throughout the growing season. Since a grower does not know when infection will occur, he must apply the fungicide throughout the growing cycle. Total reliance on fungicides for root rot control is not only expensive but also it may be unnecessary. Emphasis should always be placed on correct cultural practices as described previously rather than relying on fungicides.

However, use of fungicides in nursery operations for root rot control is important to prevent spread of *Phytophthora* from infected plants to healthy plants. Thus, if root rot symptoms should develop on a few plants in a container block, application of fungicide to all plants in the block would be recommended to prevent further spread to healthy plants. Symptomatic plants that are infected should be destroyed to prevent spread of spores.

Use of resistant cultivars can reduce incidence of Phytophthora root rot not only in the nursery but also in the landscape. Among woody ornamentals, only cultivars of hybrid rhododendron and evergreen azalea have been evaluated for resistance. This resistance can be greatly reduced by moisture stress caused by either too much or too little water.

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Nematode Diseases of Woody Ornamentals

D. M. Benson, J. T. Walker, and K. R. Barker

Several genera of plant-parasitic nematodes including *Meloidogyne* (root knot nematode), Criconemella [syn. *Macroposthonia, Criconemoides*] (ring nematode), *Pratylenchus* (lesion nematode), *Tylenchorhynchus* (stunt nematode), *Rotylenchus* (reniform nematode), *Paratrichodorus* (stubby-root nematode), and *Xiphinema* (dagger nematode) cause damage in various woody ornamentals.

Although root knot caused by *Meloidogyne* sp. was first discovered on greenhouse plants in 1855, it was 100 years before this disease was recognized on woody ornamentals. These nematodes invade susceptible roots causing large galls to develop. In contrast to these internal feeders, *Xiphinema* (dagger nematode) and *Tylenchorhynchus* (stunt nematode) feed externally but were not known to cause damage to any plant until the 1950s.

The first phase of study was to identify and describe the natural nematode communities associated with the root system of various ornamental plants. Since nematode communities are usually comprised of more than one nematode genus or species nematologists were uncertain of the nematode species that were damaging to a specific host and those nematode species that were tolerated. For instance, the root zone of a Japanese holly with nematode damage may have three or four different plant parasitic nematode species. Research has gradually developed into the quantitative area where the effects of a single nematode species at various population levels have been examined on a host by host basis. This quantitative approach has given nematologists a better understanding of nematode-host relationships.

Nematodes are found in soils throughout the southeastern United States, as well as all areas of the United States and most of the world. Nevertheless, the most damaging species of nematodes, such as *Meloidogyne incognita, M. arenaria, Pratylenchus vulnus* (lesion nematode) and *Paratrichodorus minor* (pin nematode), are found in the warmer regions of the country. Typically, nematodes are more damaging on light-textured, sandy soils of the coastal plain region of the Southeast. Woody ornamentals growing in areas that experience periodic droughts are more likely to suffer from nematode damage than plants grown under adequate moisture conditions.

Foliar symptoms of nematode damage may be apparent during the first growing season, but plant decline usually occurs as nematode damage to the root system intensifies over several seasons. Foliar symptoms include chlorosis (similar to iron deficiency) and necrosis, defoliation, miniature leaves, bronzing of the foliage especially on boxwood, stem-tip dieback and overall dwarfing of plant size. Dwarfing is one of the most common symptoms of nematode damage in woody ornamentals; however, in fields with only a few nematodes, growers may fail to recognize the problem since all the plants may be slightly dwarfed. Irregular or spotty growth in a field is a primary indicator of nematode damage in large plantings reflecting the spotty distribution of nematode populations in given nurseries (Figs. 9-12).

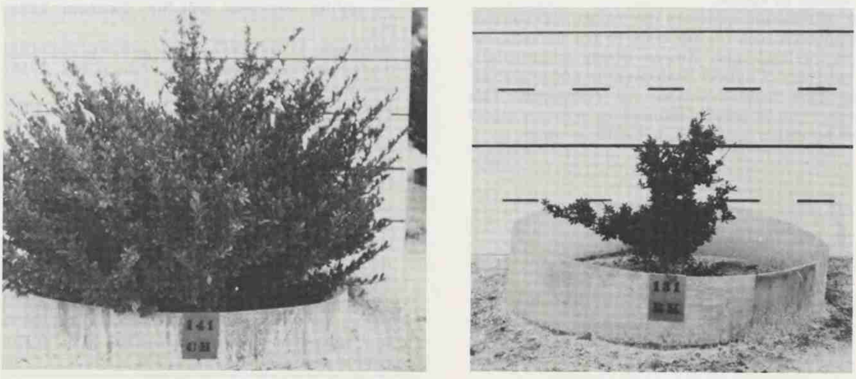

Figure 9. Japanese holly (*Ilex crenata* 'Compacta'): Healthy plant on left; plant damaged by root knot nematode (*Meloidogyne arenaria*) on right (D. M. Benson, NCSU).

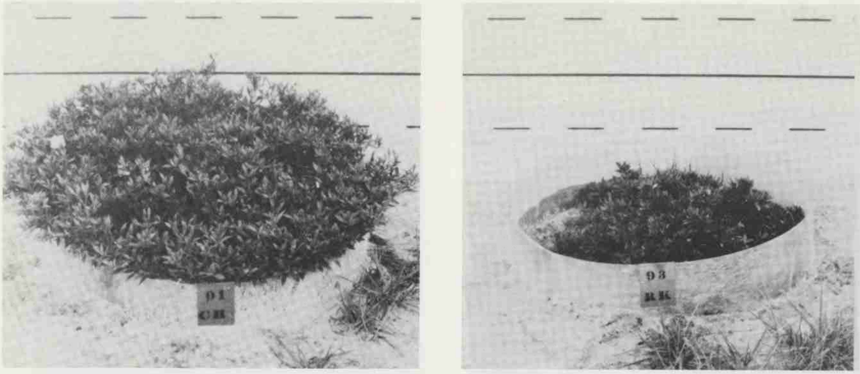

Figure 10. *Gardenia radicans*: Healthy plant on left; plant killed by root knot nematode (*Meloidogyne arenaria*) on right (D. M. Benson, NCSU).

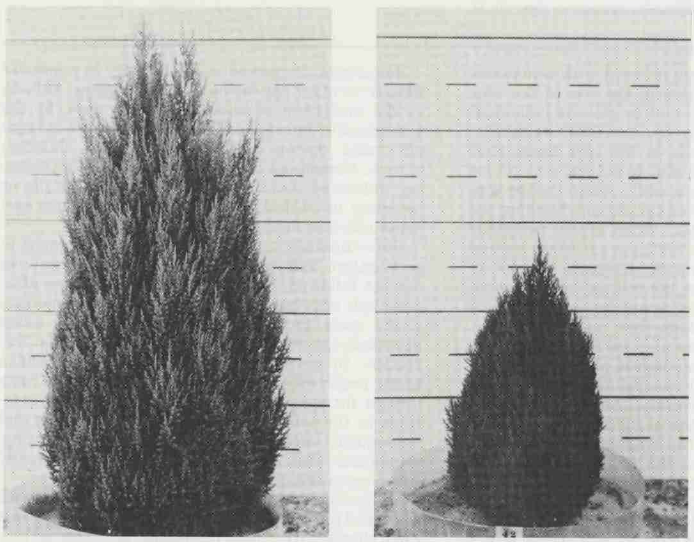

Figure 11. Spiny Greek juniper: Healthy plant on left; plant damaged by lesion nematode (*Pratylenchus vulnus*) on right (D. M. Benson, NCSU).

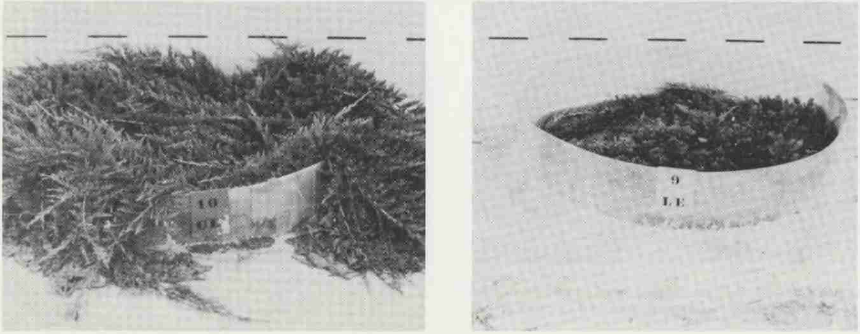

Figure 12. Blue rug juniper: Healthy plant on left; plant damaged by lesion nematode (*Pratylenchus vulnus*) on right (D. M. Benson, NCSU).

Root symptoms of plants infected with nematodes include an overall reduction in the size of the root system and necrotic areas and/or galls on individual roots. Plants attacked by the root-knot nematode, *Meloidogyne*, develop galls in the root tissue that range in size from 3 to 4 mm to several cms (Color Plate IV, 4). Root galls are easily recognized by digging up a portion of the root system and washing the roots in a bucket of water. When severe nematode damage occurs, plants do not have sufficient normal functioning roots to provide adequate uptake of nutrients and water from the soil. In this situation, nematode-infected plants may be killed during droughts when nematode-free plants survive. Nematode-damaged plants also are more susceptible to winter kill than nematode-free plants.

As indicated previously, several nematodes are important on woody ornamentals. The lesion nematode (*Pratylenchus vulnus*), the stunt nematode (*Tylenchorhynchus claytoni*), the ring nematode (*Macroposthonia xenoplax*), and the root knot nematodes (*Meloidogyne arenaria, M. incognita, M. javanica* and *M. hapla*) are known to cause damage to boxwood.

Other plant-parasitic nematodes have been associated with the root system of various woody ornamentals during survey work but their pathogenicity has yet to be demonstrated. In field studies, the spiral nematode (*Helicotylenchus dihystera*), the lance nematode (*Hoplolaimus galeatus*) and the stubby-root nematode (*Paratrichodorus minor*) reproduced but did not cause significant damage on a wide range of woody ornamentals compared to growth of plants in nematode-free soil.

Nematodes are spread in the nursery in a manner similar to other soil-borne plant pathogens, namely by the movement of infected nursery stock, by the movement of infested soil on equipment and by run-off water during heavy irrigations or thunderstorms. Nematodes also may be spread by introducing untreated plant parts such as peanut hulls or recycling untreated soil or potting media into new nematode-free media.

Unfortunately, many nematodes are increased in propagation beds where strict sanitation procedures are not followed. Plants become infected soon after nematode eggs hatch. The root knot larvae penetrate young roots and cause galls. All stages of lesion nematode can enter the roots resulting in lesion formation. In contrast, dagger nematode feeds via a small needle-like spear without entering the roots (except for spear). Temperature and moisture relations in the soil become adequate for hatching and subsequent infections each year during the spring. Conditions that favor good plant growth usually favor nematode reproduction.

Control of nematode diseases is similar in approach to control for other soil-borne diseases. Many growers have changed from field production to container production, using soilless mixes to avoid nematode problems (as well as root rot problems). Since nematodes move only short distances under their own power, movement within the nursery is associated with infested soil carried about on equipment, introduction of infected nursery stock or passive movement in run-off water. Sanitation measures designed to prevent these types of movement as well as chemical soil treatments will reduce the incidence and severity of nematode damage.

Host susceptibility to nematodes varies even among cultivars within a species. This means that each nematode-host combination must be studied singly to determine plant response. Many of the commonly grown species of woody ornamentals have been tested under field conditions (Table 6). Woody ornamentals not damaged by root knot, ring or stunt nematode included Formosa azalea, Camellia, Burfordi holly, yaupon holly, ligustrum, photinia and shore juniper. Nematode-tolerant plants should be used where nematodes have been a problem.

Table 6. Response of Several Ornamentals to Root Knot, Stunt, Lesion or Ring Nematode After 3 Years in Field Microplots.

Host Plant	Nematode Reaction			
	Root Knot	Stunt	Lesion	Ring
Azalea	T	S	O	T
Aucuba japonica	HS	S	O	S
Buxus microphylla (Japanese Boxwood)	HS	T	S	T
Buxus sempervirens (American Boxwood)	O	T	HS	O
Camellia japonica	T	T	O	O
Camellia sasanqua	T	T	O	O
Gardenia jasminoides	S	T	T	T
Gardenia radicans	HS	T	T	T
Ilex cornuta (Chinese holly)				
cv. Burfordi	T	T	O	T
cv. Rotunda	S	S	O	S
Ilex crenata (Japanese holly)				
cv. Compacta	HS	T	T	S
cv. Convexa	HS	T	O	S
cv. Helleri	HS	S	O	S
cv. Rotundifolia	HS	S	O	S
Ilex vomitoria nana (Yaupon holly)	T	T	O	T
Juniper sp.				
cv. Blue rug	T	T	HS	T
cv. Shore juniper	T	T	O	T
cv. Spiney Greek	T	T	S	T
Ligustrum (Privet)	T	T	O	T
Nandina domestica	T	T	T	T
Photinia fraseri (Red tip)	T	T	O	T
Rose	S	S	S	T

HS — Plants highly susceptible (severe stunting, branch dieback and death)
S — Plants susceptible (some stunting but plants will grow satisfactorily)
T — Plants will grow satisfactorily
O — Have not been tested

In summary, numerous species of nematodes cause extensive damage to woody ornamentals. This damage may be especially severe in light soils, warm climates, and drought conditions. These problems can be minimized by a combination of sanitation, careful selection of cultivars and use of chemical soil treatments.

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Pinewood Nematode

Suzanne Spencer and R. K. Jones

The pinewood nematode (*Bursaphelenchus xylophilis*) has been responsible for widespread losses to pines in Japan since the early 1900s. This nematode was first identified in the United States in Missouri in 1979. However, it has been found in nearly all the midwestern and eastern states, suggesting that it may be native to the United States.

The pinewood nematode has a wide host range including many species of pine that vary in susceptibility. Highly susceptible to susceptible *Pinus* spp. include: *nigra*, (Austrian); *densiflora*, (Japanese red); *thunbergii*, (Japanese black); *pinaster*, (cluster); *taeda*, (loblolly); *sylvestris*, (Scots); *virginiana*, (Virginia); and *mugo* (mugo).

More resistant species include: *banksiana*, (jack); *strobus*, (white); *pungens*, (table-mountain); *echinata*, (shortleaf); *palustris*, (longleaf); *rigida*, (pitch); *elliottii*, (slash); and *caribaea*, (slash).

Occurrence of the disease in hosts other than pine are more rare but include: atlas and deodar cedar, white and blue spruce, European larch, balsam fir and hemlock. Trees older than 5 years and Christmas trees in the Midwest appear to be more susceptible than younger trees or nursery stock. The disease has been severe in recent years in Japanese black pine being used as a seashore landscape plant in Virginia and North Carolina. This may reflect the attraction of the insect vector to older or drought stressed trees.

Symptoms

An early symptom of the disease is general wilting of the needles. As the disease progresses, yellowing of the needles occurs, followed by browning and death of the entire tree. Susceptible pines may die within 30 to 90 days after the first visible symptoms; more resistant pines take longer. These symptoms may be easily confused with those caused by bark beetles, *Fomes annosus* root rot and other problems. To identify the pinewood nematode, it is necessary to recover it from diseased wood (fresh branches or trunk borings) in a diagnostic laboratory.

The disease cycle is complex and involves the southern pine sawyer, *Monochamus titillator*, longhorned beetle vector. The female beetle is attracted to stressed, weakened or recently killed trees, on which she deposits her eggs. After hatching, beetle larvae feed in the trunk and larger branches of the tree during the growing season and overwinter there. The adult beetle emerges in late spring then flies to another tree to feed. It carries with it the nematodes, which may then leave the beetle's body and invade the tree through the feeding wound. The nematodes then multiply rapidly, causing development of the disease.

Control

The control of pinewood nematode involves sanitation and quick removal of diseased trees before emergence of beetle vectors in the spring. The wood should be burned, buried or debarked. High value trees and nursery stock can be irrigated during drought periods that stress the plants. In some cases, insecticides to control the vectors may prove useful. Avoidance of other conditions that stress the tree or are conducive to disease and insect attacks (all of which predispose pines to infestations) is also of value. Because it is thought that the pinewood nematode has been present in the United States for a very long time and that the insects that spread it do not aggressively attack healthy trees, damage due to it is not likely to increase over past levels.

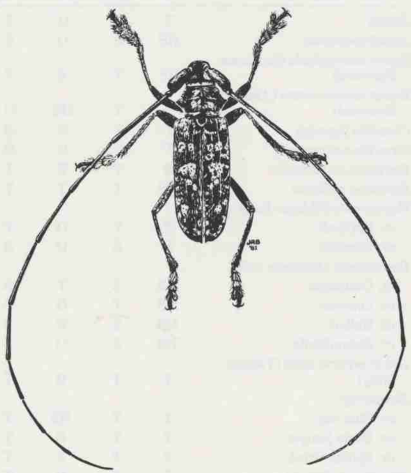

Figure 13. *Monochamus titillator*, the southern pine sawyer, the insect vector of the pine wood nematode (James Baker, NCSU).

Cylindrocladium Diseases

J. J. McRitchie

Cylindrocladium scoparium, the type species of this genus was first described in 1892 as a saprophyte rather than a pathogen. During and after the 1950s, however, the pathogenicity of this and other *Cylindrocladium* species has been demonstrated on numerous hosts, particularly woody ornamentals. Several species in this genus are now considered to be devastating pathogens capable of attacking all plant parts. They are well suited to attacking plants in propagation areas and plants pushed with high rates of fertilizer and water.

Nearly 20 species of the fungus are recognized, with a host range of approximately 100 ornamental plant species. Both hardwood and conifer nursery seedlings are particularly susceptible. Cylindrocladium leaf spots have been reported on numerous palm species as well as on such diverse woody ornamental species of *Ilex, Camellia, Leucothoe, Rhododendron, Juniperus, Eucalyptus, Cercis, Magnolia, Ligustrum*, and White pine.

The fungus may also cause serious root rots; one of the most severe occurring on azaleas. It also may cause extensive losses of cuttings in propagation houses. Cultivars of *Leucothoe*, recently introduced from the West Coast, are very susceptible to *Cylindrocladium*. Symptoms vary widely depending on the affected plant species. The fungus may cause leaf spot in holly, a stem canker or crown rot in rhododendron and rose, damping-off or root rot of red bud, a leaf spot, stem canker, root rot and quick wilt of azalea. Damping-off, needle blight, stem canker and root rot may occur in white pine.

The outer, or cortical tissues of the roots are diseased and the vascular cylinder of azalea roots becomes discolored. In some situations the vascular discoloration extends into the stem; however, discoloration does not usually appear more than an inch or so above the soil line. Latent root infections in which the fungus progresses slowly are believed to account for the late sudden wilt of plants which are several years old.

Leaf spot of azaleas has been reported from the warmer southern states and occasionally on certain varieties grown in the North. Under experimental conditions leaf spots appear 3 days after the leaves are inoculated with conidia.

It has been estimated that annual losses due to *Cylindrocladium* spp. exceed 3 million dollars. Under the proper conditions, *Cylindrocladium* spp. can be destructive pathogens to a large number of hosts. A large proportion of these hosts are ornamentals which suggests that man may be providing these proper conditions through certain propagative practices such as overhead irrigation.

The fungus *Cylindrocladium scoparium* is the cause of Cylindrocladium root rot, but also causes leaf spot and "quick wilt" of azalea. Plants are infected by conidia produced by the fungus. These conidia are disseminated by splashing water and may be wind-borne. They may splash up on the leaves or wash down to the roots, especially in a mist-rooting bed. Conidia are produced at the base of azalea plants, usually on infected leaves that have fallen to the ground.

In addition to conidia, the fungus produces weather resistant resting bodies called microsclerotia in the leaves of azaleas. It is generally believed that the fungus survives in nurseries as microsclerotia. Sclerotia have been isolated from field soil and it is believed that sclerotia in the soil germinate and penetrate the roots. Sclerotia may also produce conidia which then cause infections. Sclerotia are very resistant to soil fumigation with chemicals.

Cylindrocladium spp. caused excessive losses in nurseries and the greenhouse production of azalea cultivars during the 1960s. The disease is less commonly observed in azalea production now and this is possibly due to better recognition of the disease, better sanitation and the use of a highly effective fungicide. The disease is more common in the deep South than the upper southern states. Symptomless diseased liners are often transported and die after transplanting.

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Powdery Mildew

R. K. Jones

Plant diseases have probably been causing plant damage as long as plants have been on the earth. Powdery mildew is one disease recorded in some of man's earliest writings such as 1 Kings 8:37 and Amos 4:9.

Powdery mildew is a general name for a disease affecting many different plants (Table 7) caused by a group of fungi that reproduce similarly and produce similar symptoms. The powdery mildew fungi are classified into at least eight different commonly occurring genera with numerous species. Some of these species attack a wide variety of host plants; however, most are host specific.

Table 7. Woody Ornamental Plants that are Susceptible to Powdery Mildew.

**Cotinus coggygria*	Smoke-tree
Cotoneaster spp.	Cotoneaster
Crataegus spp.	Hawthorn
Eucalyptus spp.	Eucalyptus
**Euonymus* spp.	Euonymus
**Hydrangea* spp.	Hydrangea snowball
**Lagerstroemia indica*	Crape myrtle
**Leucothoe* spp.	Leucothoe
Ligustrum vulgare	Privet
**Lonicera* spp.	Honeysuckle
Magnolia spp.	Magnolia
**Malus* spp.	Apple, crab apple
**Photinia serrulata*	Chinese photinia
**Platanus* spp.	Sycamore
Prunus spp.	Peach, plum, apricot
Pyracantha spp.	Pyracantha
Pyrus spp.	Pear
**Quercus* spp.	Oak
Rhododendron spp.	Rhododendron, azalea
**Rose* spp.	Rose
Salix spp.	Willow
**Spirea* spp.	Spirea
**Syringa vulgaris*	Lilac
Vaccinium spp.	Blueberry
**Viburnum* spp.	Viburnum, snowball
Wisteria spp.	Wisteria

*Powdery mildew occurs commonly on this plant

Symptoms

The white, tan or gray fluffy mold or mildew appearance of infected plant parts is the characteristic symptom of powdery mildew diseases and may occur as isolated spots or cover entire leaves, stems and flowers on highly susceptible plants under environmental conditions favorable for abundant mycelial growth and spore production. Leaves of some plants affected by powdery mildew may be stunted, curled or twisted. Red pigments may also be produced in infected leaves. (Color Plate VI, 3)

Powdery mildew fungi grow mostly on the plant surface rather than inside the plant tissues as do most disease causing fungi. The powdery mildew fungi are obligate parasites and therefore grow only on live plant cells, whereas many other plant pathogens subsist on live or dead plant cells. The fungal mycelium grows on the plant surface producing asexual spores (conidia) on stalks (conidiophores). The conidia are wind borne to other susceptible plants where they germinate in high humidity and specialized nutrient absorbing structures are formed in the epidermal cells. If weather conditions are favorable, mycelium and conidia are produced to complete the cycle. Powdery mildew diseases are generally more severe in the spring and fall.

Many of the powdery mildew fungi produce sexual spores (ascospores) in the fall which may be important in winter survival on fallen leaves. The sexual stage appears as tiny black pepper-like spots in the white mycelium. Other powdery mildew fungi may overwinter as dormant mycelium on stems and buds of deciduous plants. Powdery mildew fungi are destructive on evergreen plants such as *Photinia serrulata* and *Euonymus* spp., remaining active during much of the year in southern areas.

Leaves, young stems, buds and flowers can be infected by these fungi. Young succulent tissues are more susceptible than older hardened-off tissues. Growing conditions that increase succulence can increase the severity of powdery mildew.

Powdery mildew damages plants by reducing the asthetic value of the plants, by reducing growth rate (stunting), killing buds, destroying flowers and killing new growth. Powdery mildew fungi have very high demands for nutrition and energy necessary for their growth and spore production. This must be obtained from the host plant, thus weakening and reducing host plant growth.

Epidermal cells of some plants are not compatible with the powdery mildew fungi and die when fungal penetration occurs. This produces small flecks of necrotic (dead) cells and is called a hypersensitive reaction. Since these fungi are obligate parasites and cannot live on dead cells, the hypersensitive reaction stops further disease development. This has been reported for some cultivars of rhododendron and azalea.

Control

Control of powdery mildew generally is not too difficult. Several fungicides are highly effective against these fungi. Several properly timed and applied applications can prevent new infections or eradicate the established ones. Complete coverage of all plant surfaces is necessary for good control.

Powdery mildew resistant cultivars or species of woody ornamental plants are available and offer an excellent means of control. Resistant cultivars of crab apple, lilac, crape myrtle and others are available and should be more widely grown in southeastern nurseries. *Photinia glabra* and *P. fraseri* have replaced *P.*

serrulata in the nursery and landscape trade largely because of resistance to powdery mildew.

Beneficial management practices such as improving the air movement around the plants and increasing the amount of sunlight can often reduce disease severity. Plants left in overwintering greenhouses too late in the spring can be severely damaged by powdery mildew.

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Crown Gall

G. H. Lacy and H. E. Reed

Crown gall affects wide numbers and diverse kinds of plants including ornamentals. This disease is especially important on fruit trees, brambles and certain ornamental crops. In 1976, for only 10 states reporting, crop losses to crown gall were estimated to be 23 million dollars. In California, Oregon and Washington, it is responsible for the destruction of 10 percent of nursery grown fruit trees. Numerous woody ornamentals, such as cypress, euonymous, forsythia, hibiscus, lilac, flowering peach, privet, rose, viburnum and willow are affected. Affected plants are damaged by galls or plant tumors that interfere with water and nutrient transport and result in unthrifty plants, wilting or even death. Ornamentals with galls are worthless and must be destroyed.

Symptoms

Galls are often located at the crown (soil line), but may also be found on branches, roots and, occasionally, leaves of plants (Color Plate VIII, 5). They range in size from one-fourth of an inch to several inches in diameter. Gall tissue is disorganized and develops from abnormally rapid cell division and enlargement and is susceptible to rots caused by secondary bacterial and fungal pathogens.

Crown gall was first associated with the bacterium *Agrobacterium tumefaciens* in 1907. Only in the past decade, was it discovered that the gall or tumor inducing principle (TIP) is part of a separate genetic entity, a plasmid, that is itself parasitic within pathogenic strains of the bacterium and absent in nonpathogenic strains. The "pathogenic" bacterium, then, is just a vehicle for transmission of the TIP to plants. After the bacterium attaches to plant wounds and multiplies briefly among parenchymatous cells, the TIP moves into the plant cell and is maintained with the host's genetic material. Since the vehicle bacterium is not necessary for tumor development, many galls become "aseptic" or free of *A. tumefaciens*.

The significance of a bacterium-borne plasmid being able to insert genetic material into plants has not been lost to plant breeders. Preliminary experiments have begun to establish gene engineering techniques to use this gene insertion mechanism to introduce beneficial genes into plants.

To complicate the situation further, plasmids bearing TIP may mediate their own transfer to related non-pathogenic soil bacteria such as *A. radiobacter*, a saprophyte, and *Rhizobium* spp., associated with symbiotic nitrogen fixation. Since these non-pathogens, once they have acquired TIP, may also cause crown gall, clearly *A. tumefaciens* is not the pathogen but just the vehicle for TIP. Therefore, TIP is the first example of a new class of genetic plant pathogens.

Control

Because *A. tumefaciens* and other hosts of pathogenic TIP are soil-borne bacteria, crown gall is associated with and spread by infested soil as well as infected plants. In nurseries and greenhouses, this combination of soil and plant complicates disease control. Strict sanitation is necessary to prevent spreading pathogenic bacteria during vegetative propagation that requires plant cuttings or grafting. It is imperative that stock plants be free of crown gall. However, latent infections of apparently symptomless plants can provide pathogenic inocula adequate for epidemics of crown gall. Soil used for container plant culture must be disinfected with heat or chemicals and protected from any contact with possibly contaminated surfaces such as potting benches, containers or floors. Since bacteria may also be spread by water or insects, bacteria-free water sources and chemical insect control measures must also be used routinely.

A new biological disease control technique shows promise for reducing the amount of crown gall in nursery plantings. Dipping or spraying seedlings and cuttings in suspensions of certain strains of *Agrobacterium radiobacter* before planting has given good control of crown gall. This bacterium competes with *A. tumefaciens* both as a soil saprophyte and for attachment to wound sites. The strains that have been found most useful for biological control also produce a specific toxin, agrocin 84, active against strains of *A. tumefaciens* carrying TIP bearing plasmids. The Environmental Protection Agency has approved the use of this biocontrol agent and it is available commercially as Galltrol A® or Norbac 84® for control of crown gall on non-bearing almond, apricot, cherry, nectarine, peach, plum, prune, raspberry and walnut, as well as ornamentals such as euonymus, rose and weeping cherry.

Some knowledge about biological control agents is necessary in order to obtain the most effective use of this control technique. For example, TIP bearing strains of *A. tumefaciens* are naturally resistant to agrocin 84. Susceptible strains may become resistant either through mutation or acquisition of plasmid borne agrocin 84 resistance, and agrocin 84 does not prevent above ground gall formation. Therefore, pathogenic bacterial strains isolated from crown gall on specific crops should be tested in the laboratory for their susceptibility to agrocin 84 and sanitation must be used to prevent infections of above ground plant tissues.

Southern Blight—*Sclerotium rolfsii*

Charles Hadden

Southern blight, caused by the fungus *Sclerotium rolfsii*, has been identified on hundreds of different plants for many years. Southern blight affects plants from many different plant groups including field crops and vegetables as well as ornamentals. Hundreds of different herbaceous ornamentals and numerous woody nursery crops are also affected (Table 8). Woody plants are susceptible to southern blight in the seedling stage and for several additional years until a thick bark is formed on the lower stem or trunk. Aucuba appears to be susceptible for many years, however.

The first sign of the disease is the formation of white mats of mycelium at the base of the plant stems (Color Plate V, 5). This spreads upward in somewhat of a fan shape and sometimes spreads out 1 or 2 inches over the ground around the plant stem during warm wet weather. The round sclerotia formed in the mycelia mats are at first white but later turn tan to light brown and may be as large as 1/16 of an inch in diameter (Color Plate V, 7). At times they may be numerous enough to form a light crust over the soil for several inches around the stem of the infected plant. While the sclerotia are in the white stage, droplets of liquid often form on the sclerotia. The fungus produces a toxin, oxalic acid, that kills plant cells in advance of the mycelial growth. Because of this, the pathogen never penetrates living tissue, which explains why so many different kinds of plants are readily affected by southern blight.

In the nursery environment, the disease can be serious when proper sanitation procedures are not carried out. It is most likely to occur on field grown plants during the summer months. When southern stem blight is detected in container ornamentals, the affected plants and soil should be carefully removed and destroyed. Disease occurrence is usually spotty in container plants. Care should be taken that none of the potting mix or soil be scattered or dropped in the nursery since the hard sclerotia are resistant to fungicides and soil fumigants. Fungicides are effective only after the sclerotia have germinated but before host penetration occurs.

When the fungus attacks field-grown ornamentals, carefully remove the plants and soil around the plants and discard. When large areas of the field are affected, fumigation and/or crop rotation may be required.

Table 8. Some Woody Ornamental Plants Attacked by *Sclerotium rolfsii*.

Ajuga reptans	Bugleweed
Althea rosea	Hollyhock
Aucuba japonica	Gold dust plant
Camellia sinensis	Tea
Crytomeria japonica	Crytomeria
Cydonia oblonga	Quince
Eribotrya japonica	Loquat
Hydrangea macrophylla	Hydrangea
Juglans nigra	Black walnut
Malus species	Crab apple and apple
Persea americana	Avocado
Pittosporium species	Pittosporum
Prunus persica	Peach

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Web Blight—*Rhizoctonia solani*

R. K. Jones

Web blight or Rhizoctonia foliar blight is caused by the very widespread fungus *Rhizoctonia solani*. This disease usually develops in the field during July, August and early September, but the damage may still be visible on diseased plants in the fall and winter. Web blight rarely kills woody ornamentals but drastically reduces plant quality. The disease first appears as a dark brown spot or blight on the lower inner leaves. Under warm, moist conditions, leaf blighting develops rapidly, often starting along the leaf margin. Blighted foliage is webbed to the stems by the fungal growth (Color Plate VI, 5). Another phase of web blight occurs when cuttings are taken from diseased plants in the field. Foliar blight and death of the cuttings then occurs during propagation, particularly under mist. Losses during propagation have at times been extensive and misdiagnosed.

Web blight develops in tight blocks of containerized plants. A thick, dense canopy of leaves that prevents drying of leaves in the center of the plant favors disease development. The fungus grows up from the medium in the container to blight the lower leaves. Disease development is favored by warm, humid weather, high nitrogen rates, shade, succulent plants, low air movement, crowded plants and frequent irrigation. Under favorable conditions, the disease develops rapidly causing extensive damage, even killing small plants.

Web blight was first observed on azaleas in Florida in 1960. It is commonly observed on numerous plants in the nursery including Helleri holly, gray Santolina, English ivy, Yaupon holly, sheared azaleas and arbor vitae. It is more likely to occur on compact growing varieties such as Gumpo azaleas or plants pruned to maintain a compact form rather than on loose, open, leggy-growing varieties. It has also been observed on numerous species of plants in shipping boxes and poorly ventilated trucks during shipment in the summer months.

Web blight must be prevented before all of the lower leaves are killed. The disease is easy to control by improving the air movement around the plants, spreading the containers further apart, reducing rate of nitrogen and reducing the frequency and/or amount of irrigation water. If the above steps are not taken, fungicides that are effective against *Rhizoctonia* will prevent web blight if used on a regular spray schedule every 7 to 10 days during summer months. During summer months, plants should be sprayed before cuttings are taken for propagation.

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Botrytis Gray Mold of Ornamentals

R. C. Lambe

Gray mold caused by the fungus *Botrytis cinerea* is a common disease of many flowering plants and can be a problem on some woody ornamentals. The fungus causes spots on flowers and may rot emerging flowers and leaves. Woody ornamentals that are affected during nursery production are euonymus, aucuba and azalea.

Symptoms

The fungus produces masses of spores on the flowers and leaves of aucuba in the spring when the day temperature is around 62°F (17°C) and the minimum at night drops to 55°F (13°C). The fungus is capable of growth from about 28°F (-2°C) to 90°F (32°C). Mist propagation, greenhouse culture, or storage of plants in milky white plastic structures for winter protection is particularly hazardous for disease development because the temperature and humidity range is favorable in the spring. Euonymus liners that are sheared to shape the tops become covered with gray mold (Color Plate V, 4). Azaleas that are stored at low temperatures are damaged. Azalea cuttings stuck with flower buds can develop gray mold when the flowers open the following spring if kept in a greenhouse with high humidity (Color Plate V, 3).

Free moisture in the form of liquid water is necessary for germination of *Botrytis cinerea* spores. If the humidity drops, the fungus will not grow within the plant tissues; but growth will resume when the tissues become moist.

In Virginia, gray mold is most prevalent during the spring when the humidity is high. If dead or dying plant material, such as sheared leaf fragments, are available for fungus colonization, the air will become full of Botrytis spores.

Botrytis cinerea does not usually invade healthy green tissue, such as leaves and stems, unless there is injured tissue or the fungus grows from dead tissue such as fallen dead petals or leaves. The fungus grows on the dead tissue and then moves to healthy tissues. Flower petals of ornamentals are the most susceptible to Botrytis infection. Under conditions of high relative humidity, the fungus sporulates on infected tissues and produces a mass of characteristic gray or brownish spores which become airborne to other susceptible tissue.

Control

The best method of disease prevention is sanitation. Because large numbers of airborne spores are produced on decaying vegetation, including leaves, stems and flowers, it is important to eliminate or reduce the sources of spores. Therefore, all tissue such as prunings, disbuds and petals, should be picked up and removed at least daily. Dispose of this debris by burying, burning or otherwise destroying to prevent a buildup of spores. Refrigeration at temperatures near 32°F (0°C) will retard but not stop the development of gray mold.

Emphasis should be placed on environmental manipulation to prevent condensation of water on susceptible plant parts. Do not water overhead during blooming because wet flowers are highly susceptible. When necessary, watering should be done during midday so the foliage can dry as rapidly as possible. Warm air will hold more moisture than will cold air. If the relative humidity is high in the greenhouse, a drop in temperature to below the dew point will result in the formation of free moisture on cool surfaces. This condensation can be prevented by keeping the plants warm, by air movement and/or by heating the air. Because of high energy costs, growers have been attempting to reduce heating by keeping greenhouse vents closed. However, it may be necessary under some conditions to slightly open the vents to allow some of the moisture-laden air to escape into the atmosphere.

Fungicides effective against Botrytis are numerous, but the number that are registered for use on ornamentals is very limited. There are some chemicals that are injurious to the flowers and leaves. All of the fungicides are protectants and must be applied before infection. If the lower leaves of crowded plants become infected, the spores that are produced will serve as a reservoir for initiating new infections. Therefore, it is important to apply fungicides to the foliage at an early stage so that the lower foliage can be adequately covered with the fungicide. Use of chemicals varies from state to state and growers should refer to their respective state extension service recommendations.

Tolerance of Fungicides

Systemic fungicides have been successfully used to prevent gray mold, but tolerance of the fungus to this fungicide has become common with repeated applications of the fungicide. Several new fungicides have been synthesized and successfully tested. There are certain disadvantages, including unsightly residue on the leaves. However, it is possible to minimize the residues by starting a preventive program when the plants are small and then reducing the concentration of fungicides when the plants are close to saleable size. It may also be possible to reduce the rate of fungicide and still obtain protection. Thermal dusts or fumigant fungicide applications can also be effective without leaving an unsightly residue.

Additional Literature

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Damping-off in Seed Beds

R. K. Jones

Damping-off is a term used to describe a common disease that occurs on seedlings of many woody ornamentals, trees, flowers or vegetables. It results in extensive losses to growers through the production of low-quality diseased plants that grow slowly in the field or containers after being transplanted. Such plants may die of root rot months or years later. The disease can be avoided and controlled by practicing effective pre-plant control measures.

Cause

Damping-off is caused primarily by fungi in these genera: *Rhizoctonia, Pythium, Fusarium, Phytophthora, Sclerotium* and others. Any of these fungi may be present in nursery soils and seed beds. Germinating seed and seedlings, especially weak or slow growing ones, are vulnerable to attack by these fungi during periods of unfavorable growing conditions. Species of *Pythium* and *Phytophthora* are more likely to cause damping-off in cool or warm wet soils; whereas *Rhizoctonia* spp., *Fusarium* spp. and *Sclerotium rolfsii* may cause damping-off under warmer and drier conditions.

Fungal damping-off may be confused with plant injury caused by excessive fertilization, high soluble salts, drowning in wet soil, desiccation in dry soil and death of seedlings from excessive heat, cold, flue fumes or chemical injury.

Symptoms

Typical symptoms of damping-off are rotting stems at or near the soil line and root decay (Color Plant VII, 1). Affected areas in the seed bed are usually a foot or more in diameter with brown, shriveled, collapsed or stunted seedlings. Fungal Mycelium may be seen on affected plants. Germinating seed can also be attacked by these fungi before they emerge from the soil, resulting in poor stands. Some fungi that cause damping-off may be seed-borne, but most are soil-borne.

Control

The best control of damping-off is to avoid it altogether. Once damping-off has started in a plant bed or seedling flat, it may be difficult to control. The following methods are employed to prevent damping-off: 1) proper soil preparation and management to provide for good drainage, structure, aeration, water-holding capacity and plant nutrition by including proper amounts of fertilizer and lime according to the soil test report; 2) proper soil treatment to reduce the level of fungi that cause damping-off; 3) the use of fungicide-treated seed with high germination (specify treated seed before purchasing); 4) proper seeding rates to avoid thick plant stands, poor air movement and low light intensity; and 5) strict sanitation to avoid reinfesting treated soil with pathogens. Many outbreaks of damping-off can be attributed to poor sanitation practices after treating the soil. For small nurseries, it may be practical to buy sterilized media and eliminate steps 1 and 2 above.

Once damping-off has started in a bed or flat, it may be controlled by providing drier conditions for seedling growth. This can be done by increasing greenhouse temperatures, increasing air circulation and ventilation, reducing the frequency of watering, providing better water drainage by ditching inside and outside the plant bed structure, and by increasing the amount of light by removing such things as dirty covers and overhanging branches.

If the above preventive control measures fail, several fungicides are available that may be effective if applied as a drench or heavy spray as soon as first symptoms of damping-off are observed and by providing proper growing conditions. Since the fungi that cause damping-off are quite diverse, no one fungicide will control all of them. Therefore, combinations of fungicides are usually necessary until the pathogen is identified. Rapid identification of the causal fungus should be obtained so that proper chemicals can be applied. (See your county agent or disease clinic for diagnosis.) Several applications of the fungicide may be necessary. Check the label carefully to use the proper fungicide and rate for a particular crop.

Important Diseases of Major Crops

Diseases of the most important woody ornamental crops grown in nurseries in the Southeast are discussed in this section.

Aucuba Diseases

Kenneth Whitam

Aucuba japonica, commonly called the Gold-dust plant, is used in landscape and container planting throughout the South. The aucuba is better adapted to shaded or partially shaded areas than most shrubs. However, shade may also enhance the occurrence of pathogens after the shrubs are sold to homeowners. Growers should be aware of the more common diseases affecting aucuba plants.

Foliage Diseases

Foliage spots are primarily caused by two species of fungi. Leaf spot caused by *Phyllostica aucubae* and wither tip caused by *Colletotrichum gloeosporioides* produce a similar problem on the leaves. The spots are usually quite large and nearly black, affecting one-third to one-half of the leaf. On examination, small black fruiting bodies can be observed in the infected tissue. If these diseases are allowed to go unchecked, a tip dieback (wither tip) may occur especially from *C. gloeosporioides*. Routine fungicidal sprays will prevent foliage diseases in aucuba nursery stock.

Environmental problems may also be reflected in the foliage. Necrosis of leaves at the tips and margins is indicative of a root or soil problem caused by overwatering. Obviously, watering problems can be prevented by adjusting the watering schedule or the drainage of growth medium.

Plants exposed to cold nights and bright sun in the early spring may die back suddenly. If temperatures are sufficiently cold, aucuba may experience winter injury. Injury of this type can be prevented by protecting plants during the winter and early spring months. If damage occurs, quickly prune out dead branches to prevent fungi from invading the dead tissue. The leaf spot fungi discussed above rapidly invade leaf tissue damaged by the sun or low temperatures.

Root and crown rots caused by soil-borne fungi are the most serious diseases of aucuba in nurseries. Root rot diseases caused by *Phytophthora cinnamomi* and *P. citricola* are more serious on plants grown in poorly drained media and plants that are overwatered. Plants eventually wilt and die if the disease is not controlled (Fig. 14). Even though the death of a plant may appear suddenly, chances are chronic injury has been occurring for some time. Small feeder roots are attacked first, gradually moving into larger roots which turn black and become mushy. Growers should periodically check containers for dark decaying roots. Soil drenches are available that specifically control root rots and should be used routinely.

Aucuba is also susceptible to the southern stem blight disease caused by the fungus *Sclerotium rolfsii*. Infected plants wilt suddenly and die (Color Plant V, 6). The pathogen can usually be seen at the base of the wilted plants as white strand of the fungus plus white sclerotia that later turn brown (Color Plate V, 7). The disease occurs sporadically in container production, seldom killing more than a few plants. Remove and discard diseased plants and soil. For more information see Southern blight (*Sclerotium rolfsii*) under General Diseases.

Several nematodes attack aucubas. Root-knot nematode causes easily seen galls to develop on the roots. However, some forms of nematode damage are not as evident and can only be detected by submitting a sample to the state Nematode Advisory Service. Additional information on nematodes is given in the general section.

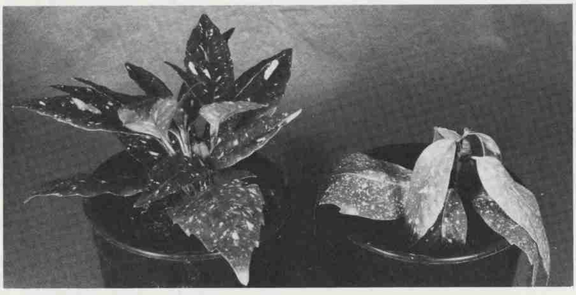

Figure 14. Phytophthora root rot of aucuba (right) and healthy aucuba (left) (R. C. Lambe, VPI & SU).

Azalea Diseases

R. C. Lambe, R. K. Jones, W. H. Wills and D. M. Benson

Azaleas are one of the most important woody ornamentals grown and are produced commercially both in the field and in containers. Production in the field is popular in some middle Atlantic states like Virginia and Maryland. However, production in containers is the most common method in the southern United States. The two different methods of production favor certain important diseases of roots and foliage. Interstate movement of diseased azaleas has been a principal problem for growers because diseased but symptomless plants are easily overlooked during plant inspection.

In the past growers have failed to practice adequate sanitation and cultural disease prevention in propagation. As a result not only have plant losses due to disease occurred in propagation, but diseased liners have been moved to the field or into containers resulting in heavy losses at later dates. Fungicides have been applied to compensate for this lack of sanitation.

Root rots caused by species of *Phytophthora, Pythium, Cylindrocladium,* and *Rhizoctonia solani* have been troublesome, especially where propagation is attempted with previously used rooting media, with containers or flats resting on the ground and with misting with water from ponds used to catch nursery run-off water. Foliage blights and leaf spots develop if cuttings are taken from diseased stock plants or if misting is so prolonged that cuttings never dry off. If the rooting medium, consisting of fine particles or peat moss, is kept wet so that it becomes soggy, rooting will be poor and cuttings will be colonized by various fungi. Application of fungicides over cuttings contrary to label directions will injure leaves and stems predisposing them to fungus infection. Rooting media that is contaminated, for example, media that is used a second time for propagation with no disinfesting treatment, may be infested with pathogenic fungi which will colonize cuttings. Propagation on or near the ground increases the chance that soil containing pathogens will splash onto the cuttings resulting in disease. Containers, flats and benches used in propagation should be new or disinfested. Fungicides can be applied to rooted cuttings to prevent root infection.

Fields destined to be planted to azaleas are usually fumigated before planting to eliminate soil-borne pathogenic fungi and nematodes. Although the cost of fumigation is high (900 to 1000 dollars per acre), elimination of *Phytophthora* spp. parasitic nematodes and weed seed is believed to pay for the expense of fumigation.

Powdery Mildew

Occasionally evergreen azaleas are infected with powdery mildew caused by either of the fungi *Erysiphe polygoni* or *Microsphaera penicillata,* but more frequently certain deciduous azalea cultivars are infected. Cloudy overcast weather conditions, in the field or in the greenhouse, are frequently favorable for mildew development. Symptoms on young leaves first appear as white powdery spots. With continued favorable environment, the fungus covers the entire lower or upper leaf surface.

Mildew spores in the white powder are carried by wind or mechanical action to other new leaf tissue where they infect and cause additional disease. It has been demonstrated that powdery mildew from azalea can infect rhododendron and vice versa. Mildew developed more rapidly at warm temperatures (25°C day/15°C night) than at cool temperatures (20°C day/10°C night) but the damage was more severe at the lower temperature. Young foliage was more susceptible than the old. Plants grown in greenhouses, under shade or with high rates of nitrogen fertilizer are more susceptible.

The overwintering perfect stage of the fungus called *Cleistothecia,* form on deciduous azaleas but their role in fungus survival has not been researched. Dormant mycelium in infected buds may be important to overwintering of the fungi. The disease has seldom caused significant damage on evergreen azaleas. Some deciduous azalea cultivars appear to have some resistance and offer the best method of control.

Leaf and Flower Gall

Leaf and flower buds are infected by the fungus *Exobasidium vaccinii* which causes galls to develop. Under very humid conditions as may occur under glass culture, the galls may become so abundant as to cause considerable harm to plants if some control measures are not implemented.

Closely related species of the fungus *Exobasidium* cause the same type of gall formation on plants such as Arbutus, Blueberry, Camellia, Ledum, Leucothoe and Rhododendron. The disease causes the leaves to become swollen, curled and form fleshy galls (Color Plate VII, 2). Galls are pale green to white or pink in color during the early stages of the disease and turn brown and hard as the season progresses. Infected flowers are fleshy, waxy and swollen. Galls are made up of abnormal tissue. Lower leaves on plants are usually the most seriously damaged portion, but under humid conditions and in shaded locations, galls may occur at the ends of top branches. The galls become covered with a whitish mold-like growth during periods of high humidity.

The occurrence and intensity of the disease is dependent upon weather conditions and upon a source of the fungal spores. The spores produced in the whitish mold on the surface of the galls are blown and washed to leaf and flower buds causing infections.

Where only a few plants are involved, as in a home planting or a small greenhouse area, the disease is kept in check by picking the galls and destroying them. Fungicides applied to the developing leaves and flowers will usually prevent infection. Some evergreen varieties are less susceptible than others.

Petal Blight

Petal blight caused by the fungus *Ovulinia azaleae* is a serious disease of azaleas in gardens. It occurs on container azaleas in nurseries and although it is not considered important because growers are primarily concerned with the foliage, this is believed to be a major source of disease in home gardens. Indian and Kurume type azaleas are severely affected, but all other azaleas and some rhododendrons are also susceptible. If the environmental conditions are favorable, the disease may spread so rapidly as to completely destroy flowers in 2 to 4 days.

Petal blight occurs primarily on out-of-door cultivars in the warmer regions of the southern United States but has been reported occurring out-of-doors as far north as Connecticut. In northern states, sclerotia of the fungus may not survive the low temperatures and may require introduction of infected plants each year. The disease also attacks azaleas in greenhouses.

Spots on the petals are first apparent when they are about the size of a pinhead. They are pale or whitish on colored flowers and rust-colored on white flowers (Color Plate V, 1). At first they are circular, but then they enlarge rapidly into irregular blotches with the affected tissue becoming soft and disorganized. Eventually, the entire corolla collapses. Infected petals are slimy and fall apart readily if rubbed gently between the fingers. This test distinguishes diseased flowers from those injured by low temperature, insects or other causes. Diseased flowers dry and cling to the plants for some time, presenting an unsightly appearance whereas healthy flowers of Indian azaleas fall from the plant while still displaying color and normal shape.

Ovulinia azaleae produces hard, black objects known as sclerotia in the blighted flowers (Color Plate V, 2) (Fig. 15). Small tan, cup-shaped reproductive structures called apothecia develop from sclerotia on the soil surface in the spring (Fig. 16). Spores are propelled to flower buds initiating primary infections. Secondary spores are produced in large numbers on the infected petals. The wind-borne secondary spores are responsible for widespread outbreaks of flower blight (Fig. 17). Under greenhouse environment, an abundance of spores is produced on infected petals. Sclerotia produced in diseased petals drop to the ground and remain undetected. Unsold, container-grown azaleas carrying sclerotia may be held over for forcing again the following year and serve as a source of primary inoculum for the disease. Infested containerized azaleas sold and set in the landscape carry the sclerotia to provide primary inoculum for future disease epidemics.

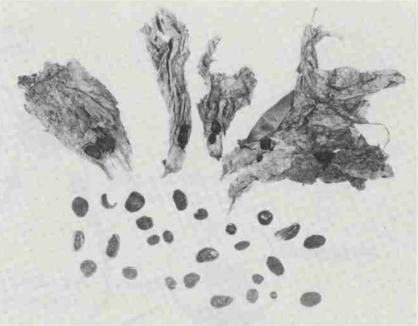

Figure 15. Black sclerotia, the survival structure of the azalea petal blight fungus (*Ovulinia azaleae*). Bottom sclerotia are separated from the blighted petal tissues (Linderman and Coyier, USDA, Corvallis, OR).

Figure 16. Apothecia, sexual spore producing structure, of the azalea petal blight fungus (*Ovulinia azaleae*) growing from the black sclerotia. The primary inoculum to initiate disease development is produced in the apothecium. The background material is sand (Linderman and Coyier, USDA, Corvallis, OR).

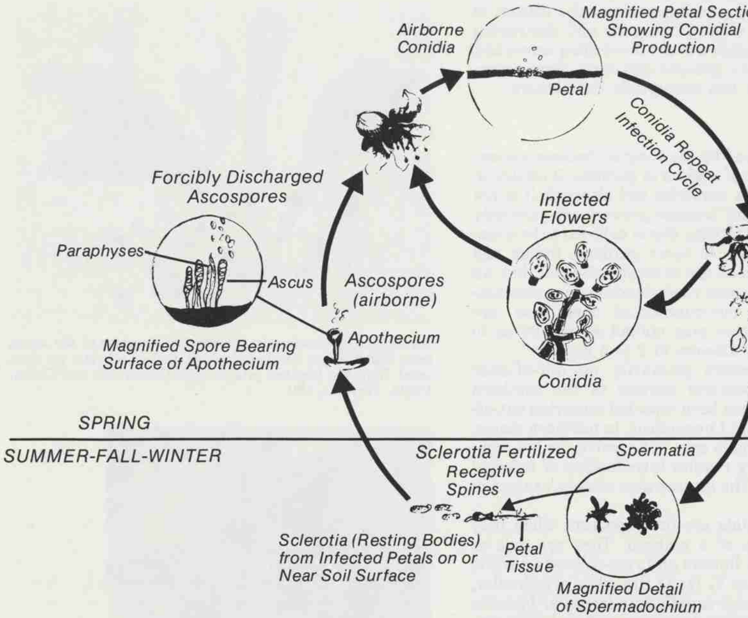

Figure 17. Azalea petal blight disease cycle (Linderman and Coyier, USDA, Corvallis, OR).

Picking and destroying affected flowers and replacing the surface litter under infected plants with uncontaminated material, are means of reducing the sources of primary infection in the spring. Fungicides will protect the flower buds from infection if they are applied when the buds start to show color.

Phomopsis Dieback

Under environmental stress conditions such as drought and during production in the field, azaleas in the landscape and stock plants become susceptible to infection by the fungus *Phomopsis* sp. resulting in stem dieback. This disease is more common in large landscape plants than in small plants in the commercial production fields. The primary symptoms of dieback are death of leaves and stems on portions of the top and a reddish-brown discoloration of the wood in diseased stems (Fig. 18). Diseased stems remain attached to the plant. *Phomopsis* sp. is chiefly a wound pathogen affecting stem tissue. Stem wounds up to 8 days old are susceptible. After infection of stems, living stem tissue is progressively killed and eventually entire branches may be killed. Pruning wounds are probably the most important infection sites. Preventing moisture stress and stem splitting from cold injury are important control methods.

Figure 18. *Phomopsis* sp. dieback on azalea stem. Bark has been removed to expose dark discoloration which is characteristic of this disease (R. K. Jones, NCSU).

Cylindrocladium Blight and Root Rot

The fungus *Cylindrocladium scoparium* attacks the leaves, stems and roots of evergreen azaleas. Cuttings become infected during propagation when they are collected from stock plants infected with *Cylindrocladium* leaf spot. During rooting under mist, *Cylindrocladium* conidia from infected cuttings are dispersed to healthy cuttings. Symptomless but infected cuttings spread the disease. Infected azalea leaves fall to the surface and provide conidial inoculum necessary to infect roots. The disease is most severe under humid conditions and often appears as a leaf spot, but diseased plants may show root rot and/or a sudden wilting of the top. It has been reported that the wilt phase of the disease is aggravated in plants subjected to overwatering, overfertilizing, high salts or other stress factors.

Disease-free cuttings should be placed in rooting medium that is free of the fungus pathogen. Healthy cuttings can be collected from clean stock plants. If the rooting medium is to be used again, it should be sterilized after removing the crop of rooted cuttings.

Web Blight

Rhizoctonia solani is the causal pathogen of leaf and stem blighting of evergreen azaleas. Very rapid symptom development occurs, characterized by small necrotic areas on leaves which are initially tan and irregular in outline. These enlarge rapidly and become dark brown to almost black. Typically the lesions advance along the margin, inward toward the midrib until most of the leaf is necrotic by the time abscission occurs. This disease is very damaging under humid high temperature propagation causing up to 100 percent defoliation. Plants grown in containers are especially susceptible where the plants are crowded together and irrigated by overhead sprinkler irrigation. Container plants are more susceptible than field grown ones. Crowding for cold protection in the fall or failure to space the plants early enough in the spring can result in serious disease development to the plant centers, especially to the lower stems and leaves of the plants. Culture in ground pine bark growing media on crowned beds does not insure against infection by *Rhizoctonia* as it does with some root rots. Blighted leaves on container plants usually remain attached to the stems. Irrigation water should be applied through sprinklers in the morning or midday so that the foliage will dry quickly. Evening irrigation so that the plants are wet at night will provide a favorable environment for disease. Application of fungicides to the foliage will protect against infection. Proper spacing of container grown plants so that they do not become crowded will often avoid disease.

Botrytis Blight or Gray Mold

During propagation in the spring, temperature and moisture may become favorable for Botrytis gray mold caused by the fungus *Botrytis cinerea* to develop on the cuttings. On cuttings that bloom, the flowers often become infected and large numbers of spores are produced on the flowers (Color Plate V, 3). These spores will initiate leaf infections. Gray mold first appears on fully developed flowers and spreads to emerging flowers and leaves. If azaleas grown for forcing are improperly stored, they may become infected.

Botrytis is capable of growth when the temperature ranges from 28°F (-2°C) to 90°F (32°C). However, free moisture on the plant tissue in the form of liquid water is necessary for germination of *Botrytis* spores. This situation occurs when humid air in greenhouse culture is cooled to the point where the water vapor condenses into water droplets on petals and leaves. If the humidity drops, the fungus will grow only within plant tissues; but fungus sporulation on plant tissue surfaces will resume when the humidity increases sufficiently.

Botrytis does not normally invade healthy plant tissue. The fungus prefers to colonize dead or dying plant tissue, such as fallen leaves and petals. If such dead or dying plant material is left lying around the propagation area it may very quickly become colonized by the fungus. Then when the temperature and humidity are favorable, air movement will carry spores to petals. The most effective method of disease prevention is sanitation in the greenhouse. Because the fungus produces spores on dead and dying vegetation, particularly decaying leaves and flowers, it is important to eliminate these spore sources. Therefore, all plant tissue such as prunings, disbuds and fallen petals should be picked up and removed at least daily, particularly when the temperature and humidity are favorable for disease. Fungicides effective against *Botrytis* are numerous.

Anthracnose

Anthracnose caused by the fungus *Colletotrichum azalea* causes leaf spotting and defoliation of both "Indica" and "Kurume" type azaleas (Fig. 19). First symptoms of leaf spotting appear on young leaves. Mature leaves generally are not infected, but severe defoliation of young shoots can occur. After the young leaves have fallen, acervuli containing conidia of the fungus develop. Therefore, control should include removal of dead leaves from the surface of the potting medium. A fungicide applied before inoculation with the fungus has been effective in control. Anthracnose and leaf spots caused by such fungi as *Cercospora*, *Pestalottia* and others are common on senescent leaves during fall and winter months on plants weakened by root rot.

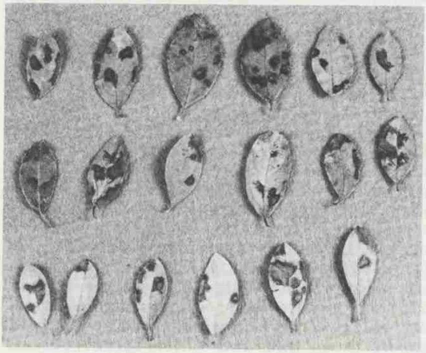

Figure 19. Anthracnose leaf spot of azalea. Several other fungi cause similar leaf spots of azalea. These diseases build up during fall and winter months and can cause severe defoliation by spring (R. C. Lambe, VPI & SU).

Salt Injury—Excess Soluble Salt

Azaleas are very sensitive to excess soluble salts. A burn on the margins of lower leaves is one symptom of excess salinity. This is followed by leaf drop. A red coloration of the leaves of some varieties of evergreen azaleas is also a symptom. Some varieties like Hexe, Lenthegruss and Vervaeneana are reported to be less salt sensitive than others. Such injury can occur on one side of the container grown plants if fertilizer is deposited in one spot on the medium surface.

Phytophthora Root Rot

The single most important disease of azaleas is root rot caused by species of *Phytophthora*. The most commonly isolated pathogen is *P. cinnamomi*, a fungus of worldwide distribution and omnivorous habit, attacking more than 1900 known hosts. Several other species of Phytophthora also attack *Rhododendron* hybrids and are potential or actual pathogens of azaleas. They include *P. cactorum*, *P. citricola*, *P. cryptogea*, *P. citrophthora*, *P. lateralis*, *P. gonapodyoides* and *P. megasperma*. Only *P. cinnamomi* is considered to be of major importance.

Symptoms vary greatly with cultivar, growing conditions and time of year. Azaleas growing in containers under ideal irrigation, drainage and other cultural practices may become infected with *P. cinnamomi* but show no foliar symptoms. Symptoms on container-grown plants include chlorosis of new foliage (yellowing between the veins), low plant vigor, dwarf leaves, defoliation, dieback and decline and death (Color Plate I, 4, 5, 6 (Fig. 20). Under similar growing conditions, a healthy plant may produce 6 to 8 inches of new growth while a diseased plant may make only one-half to 1 inch of growth. Infected plants of cultivars with red flowers frequently produce excessive amounts of red pigments in the leaves in the fall and winter whereas Snow and other white flowered cultivars produce yellow leaves in the fall. Fall defoliation, which produces naked stems with a tuft of dwarf leaves around the terminal bud, is much more extensive on infected plants.

With plants grown in the field, greenhouse or in poorly drained media, the first symptom noted is slow wilting and bronzing of the foliage, but this is actually preceded by extensive rotting of the fibrous root system. Some browning of the basal portion of the stem may accompany these symptoms. Usually, the entire plant wilts at one time. Finally, the foliage turns brown and the leaves drop off. The fungus may be cultured from the brown, rotting roots on culture media selective for water molds.

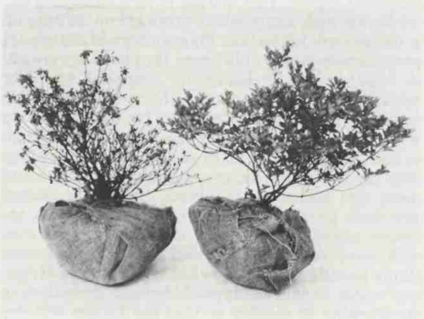

Figure 20. Phytophthora root rot of azalea (R. C. Lambe, VPI & SU).

Although *Phytophthora cinnamomi* is primarily a root pathogen attacking the small feeder roots of the host, it can colonize the larger roots and lower stems to some extent. It is able to survive long periods of time in the soil in the absence of the host. It is a difficult pathogen to control and one which is unlikely to be eradicated where it is established. This fungus produces several microscopic reproductive bodies; oospores produced in matings of compatible strains and asexually produced sporangia and chlamydospores. The sporangia produce motile spores which can move through the soil on films of water. Chlamydospores are thick-walled resting structures which serve as means of survival during periods unfavorable to active growth of the fungus.

These structures are not produced in any regular sequence making up a life cycle but are produced in response to environmental and nutritional conditions; hence, the disease caused by this fungus is one highly responsive to environmental influences. The single most important environmental factor favoring disease development is high soil moisture and soggy conditions. Relatively high soil temperatures also favor disease development.

Although *P. cinnamomi* can survive long periods in the soil the main source of inoculum for new disease epidemics is already colonized plants and the medium in which they are growing, either soil or potting mixes. Infected plants may be maintained in a state of apparent health by proper care in a nursery, particularly where the plants are subject to minimal moisture and temperature stresses. Such diseased plants easily escape detection. They may be sold and planted out in a nursery or landscape and then initiate new centers of disease. Under stressful conditions, subsequent death of the already-diseased plants is common.

Epiphytotics of root rot in azaleas may occur rapidly in nurseries when plant containers become immersed in water on top of plastic soil covers or are otherwise grown in constantly wet environments. The disease may be aggravated by recycling contaminated irrigation water from ponds or reservoirs. The soil where runoff occurs may become infested and drainage into water sources such as streams and rivers may result in more widespread disease development. Azaleas grown in the field die in large numbers from *Phytophthora* spp. when the fields are kept wet or are poorly drained.

The first line of defense in control of Phytophthora root rot is prevention through maintenance of sanitary conditions during propagation. Cuttings should be taken from healthy plants only, propagated in new or disinfested medium and after rooting transplanted to pathogen-free potting medium. At every stage of plant culture new or disinfested containers, cold frames and flats should be used. Irrigation water should come from an uncontaminated source. Benches and the ground around them should be kept free of weeds and diseased plants and watering hose sprinklers should be kept out of contact with soil.

Should *Phytophthora* spp. be cultured from plants in containers in a nursery operation, the diseased plants should be segregated and destroyed and steps taken to prevent drainage from the area of disease into irrigation sources or into uncontaminated growing and propagation areas. Good drainage is essential to prevent water-logging of the roots in the bottom of containers and can be implemented by crowned beds or crushed rock several inches thick for the containers to rest on. Chlorinated city water and well water is unlikely to be infested with viable propagules of *Phytophthora* spp. Pond or recycled irrigation water can be chlorinated at some expense, but for chlorination to be effective, water must remain in the line at least 1 minute at a residual chlorine concentration of 1 ppm. Fields intended for azaleas should be well drained and fumigated.

Until recently, registered fungicides have been only moderately effective for the control of *Phytophthora* spp. There are now several experimental systemic fungicides. A recently registered systemic fungicide can be used as a protective drench with a high level of efficacy against Phytophthora spp. This material that is taken up by the plant is systemic in action and thus provides protection throughout the plant body when used according to label directions.

The history of plant protection is a story of promising disease and insect control chemicals which for various reasons, usually pest or pathogen resistance, have had a limited useful existence. These new fungicides may also follow that pattern. The long range answer to that problem is the development of disease resistance in the host plant. Azalea varieties differ in resistance to Phytophthora root rot. When 73 varieties in 10 hybrid groups of evergreen azaleas were tested it was found that few had high levels of resistance (Table 9). They ranged from the completely susceptible Jane Spaulding to Formosa, Fakir and 18 others of rather high resistance. Of the hybrid groups, the Indians had the generally highest levels of resistance among old recognized groups and the Kurumes the least. The most susceptible were a group developed at North Carolina State University. Some of the popular and commercially available cultivars, including Hershey Red, Snow, Rosebud, Coral Bells, Purple Splendour and Hino Crimson were shown to be on the low side of root rot resistance.

Table 9. Resistance of Azalea Cultivars to Root Rot Caused by *Phytophthora cinnamomi*.

Cultivar	Hybrid group[1]	Root rot rating[2]	
R. poukhanense	species	1.60	Resistant
Formosa	I	1.83	
Fakir	GD	1.90	
Corrine Murrah	BA	1.90	
Merlin	GD	2.05	
Hampton Beauty	P	2.05	
Higasa	S	2.10	
Glacier	GD	2.10	
Rose Greeley	G	2.15	
Polar Seas	GD	2.15	
Redwing	I	2.20	
Chimes	I	2.20	
Alaska	R	2.20	
New White	I	2.27	
Shin-ki-gen	S	2.30	
Rachel Cunningham	BA	2.30	
Pink Gumpo	S	2.30	
Eikan	S	2.30	
Sweetheart Supreme	P	2.35	
Pink Supreme	I	2.35	
Morning Glow	K	2.35	
R. mucronatum "Delaware Valley White"	species	2.40	Susceptible
Barbara Gail	P	2.40	
White Gumpo	S	2.40	
Rentschler's Rose	W	2.45	
Dorothy Gish	R	2.45	
White Gish	R	2.50	
Pink Hiawatha	P	2.50	
Margaret Douglas	BA	2.50	
Gaiety	GD	2.50	
Gloria	R	2.55	
Kingfisher	W	2.56	
White Christmas	W	2.60	
Sensation	P	2.60	
Prince of Orange	I	2.60	
White Jade	BA	2.70	
Copperman	GD	2.70	
Hexe	K	2.73	
Massasoit	K	2.80	
Martha Hitchcock	GD	2.80	
China Seas	G	2.80	
Warbler	W	2.85	
California Sunset	I	2.85	
Amaghasa	S	2.85	
Pride of Summerville	I	2.90	
Hinodegiri	K	2.90	
Flanders Field	P	2.90	
Robinhood	GD	3.00	Highly susceptible
Hershey Red	K	3.00	
Herbert	G	3.00	
Fortune	P	3.00	
Catawba	GD	3.00	
Marian Lee	BA	3.05	
Snow	K	3.08	
Royalty	G	3.10	
Kow-ko-ku	S	3.15	
Rosebud	G	3.20	
Mrs. G. G. Gerbing	I	3.20	
Coral Bells	K	3.20	
Treasure	GD	3.30	
Pat Kraft	BA	3.30	
Saint James	BA	3.40	
Carror	N	3.44	
Purple Splendour	G	3.50	
Pinocchio	GD	3.50	
General MacArthur	K	3.50	
Pink Pearl	K	3.60	
Johga	S	3.70	
Sunglow	N	3.80	
Hino Crimson	K	3.90	
Elaine	N	4.10	
Emily	N	4.25	
Pink Cloud	N	4.50	
Adelaide Pope	N	4.60	
Jane Spaulding	N	5.00	

[1] BA = Back Acres, G = Gable, GD = Glenn Dale, I = Indian, K = Kurume, N = NCSU, P = Pericat, R = Rutherford, S = Satsuki, W = Whitewater.

[2] 1 = healthy roots, 2 = fine roots necrotic, 3 = coarse roots necrotic, 4 = crown rot, 5 = dead plant.

Biological control is another potential future means of plant protection. In biological control, living organisms are utilized to prevent the development of a pathogen and infection of the potential host, by imposing another organism between the pathogen and its host. The mechanism of biological control may involve the preemption of a site on or in the host or chemical antagonism of the pathogen by metabolic products of the antagonist. In view of the inadequacy of genetic resistance and the uncertainty of the future of chemicals, biological control is being investigated in more and more research centers. Such control measures may be implemented in the future, either alone or in conjunction with other control measures in an integrated pest protection scheme.

Rust

Rust, caused by the fungus *Pucciniastrum myrtilli*, can be a serious disease of deciduous azaleas and the native *Rhododendron canadense*, *R. nudiflorum*, *R. viscosum* and *R. ponticum*. The alternate host is hemlock *Tsuga canadensis*.

The first symptoms appear as small circular chlorotic spots on the upper leaf surface. The fungus produces abundant yellow to orange spots on the lower leaf surface. The disease usually appears in late summer and fall. On highly susceptible deciduous azalea cultivars, lower leaf surfaces can be completely covered with spore masses and early fall defoliation can be severe.

Rust can be controlled by growing resistant cultivars (Table 10) and weekly applications of a fungicide beginning with the first appearance of the disease on susceptible cultivars.

Table 10. Deciduous Azalea Cultivar Reactions to Rust Caused by *Pucciniastrum myrtilli*.

Susceptibility Level		
High	**Moderate**	**Low**
Klondyke	Brazil	Gibralter
Peachy Keen	Clarice	Red Letter
Pink William	Exbury Crimson	Balzae
Primrose	Homebush	
Rufus	Oxydol	
Sunrise	Peach Sunset	

Nematodes

Nematodes, particularly stunt (*Tylenchorhynchus claytoni*), can cause damage to landscape and field grown azaleas. This seldom occurs in azaleas grown in containers with soilless media. Symptoms of nematode damage on azaleas are low plant vigor and lack of dark green color of foliage. Nematode problems can be avoided in azaleas by using nematode-free potting media, selecting nematode-free planting sites and preplant soil fumigation. Granular nematicides may be useful in certain situations.

Phytophthora Dieback

Phytophthora dieback or wilt of azalea caused by the fungus *Phytophthora parasitica* occurs in nurseries in Florida. Foliar symptoms appear as dark brown to black, irregularly shaped necrotic lesions. From the leaves, the pathogen invades the shoots causing dieback and eventual death of the plant. Infection of the crown and lower stem appears as defoliation of the lower stem followed by rapid wilt and death of the plant. This disease does not appear to occur widely. For more information see Phytophthora dieback of rhododendron.

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Boxwood Diseases

Wirt Wills and R. C. Lambe

One of the attractive features of boxwoods, *Buxus sempervirens* (American boxwood); *B. sempervirens* var *suffruticosa* (English boxwood); *B. microphylla* var *japonica* (Japanese boxwood) and *B. microphylla* var *koreana* (Korean boxwood), is their relative freedom from diseases and pests. Although diseases of boxwood are few, three root diseases can cause considerable loss of plants from nurseries and landscapes under circumstances favorable for disease development. These are Phytophthora root rot, nematodes and English boxwood decline. Since most boxwoods are produced under field conditions, these soil-borne diseases can be very damaging in nurseries and also may be transplanted to the landscape with balled and burlapped boxwood plants. Once field planting sites become infested with these pathogens, it may be difficult or impossible, plus costly, to grow healthy boxwoods in them again.

Phytophthora Root Rot

Phytophthora parasitica (*Phytophthora nicotianae* var *parasitica*) causes a root rot and blight on American and English boxwood. The disease was first reported from Washington, D. C. in 1933 but not studied further until 1961 when Haasis reported it from North Carolina. Since then it has been found commonly in Virginia and North Carolina and other southern states. *Buxus microphylla* and *Buxus harlandi* are also susceptible to it. Symptoms include a general root necrosis in which the roots are brown and water-soaked and the cortex is eventually sloughed from all but the large woody roots (Color Plate I, 1). Typically the basal part of the stem(s) turn chocolate brown to black for several cm above ground level (Color Plate I, 3). The foliage becomes wilted, twisted and dry. Leaf color progresses from a normal glossy dark green to grayish-green, bronze and finally brown or straw-yellow if exposed to direct sunlight, before defoliation occurs (Color Plate I, 2). Usually the foliage of the entire plant is wilted simultaneously, in contrast to the partial effects seen in the decline of English boxwood.

Infected and colonized plants may be expected to die. Disease development may be limited to plants grown in poorly drained soils in generally warmer areas of the range of boxwood. The fungus is easily cultured from decaying roots on Phytophthora-selective medium particularly during fall months. Culturing the pathogen is necessary for a firm diagnosis.

The fungal pathogen is a water mold that reproduces primarily by motile spores formed in microscopic pear-shaped sporangia which are released in the soil where they swim in films of water spreading through the root system of a plant very quickly. Irrigation practices which maintain a constantly wet environment for the roots favor this pathogen, and one of the best preventive measures is to maintain plants on well-drained sites or in well-drained media in containers where there is no standing water.

The pathogen is favored by warm temperatures. Artificially inoculated plants may die within 6 to 8 weeks; it is not known how long it takes for disease to develop in nature. A warm soil, about 85°F (30°C) and prolonged high soil moisture favor disease development.

The best control for nursery and landscape culture is to avoid wet soil conditions. Healthy, rooted cuttings put into clean potting mix in new or disinfected containers in a well-drained medium are not likely to develop Phytophthora root rot. Some systemic fungicides offer promise for chemical control when they become available for use. Do not replant in field planting sites where boxwood have previously died from Phytophthora root rot.

English Boxwood Decline

English boxwood, *B. sempervirens* var *suffruticosa* is grown principally in an area which includes eastern New York to North Carolina, roughly in a circle around the Chesapeake Bay, extending to the Appalachian Mountains, an area where it seems best adapted. In this area in the last decade, there has been a severe reoccurrence of a decline of landscape plantings of English boxwood. The symptoms superficially resemble, in individual plants, the symptoms caused by *Phytophthora parasitica* on boxwood. Distinguishing features of this problem are its restriction to English boxwood, its rather vague etiology and its geographical limitation to certain parts of Virginia, Maryland and Pennsylvania. It has been a problem in nursery fields where boxwoods have been grown for many years.

Symptoms of decline include a slow death of plants over a period of about 2 years, one branch or part of a plant dying at a time, with a resultant mosaic of foliage coloration. The foliage changes color successively from glossy dark green to dull gray-green, to bronze, to orange and to straw-yellow if exposed to full sun. Finally the foliage changes to brown. Ultimate defoliation produces a gray skeleton of branches which may persist for years if not removed. At the earliest detectible stage of foliar discoloration, advanced root decay can be detected if the plant is uprooted. A chocolate brown discoloration of the large roots and trunk extends uniformly for only a few cm above the soil line. The brown color may extend in a discontinuous pattern any distance up the main branches of the shrub. In contrast, stem discoloration caused by *P. parasitica* is continuous and limited to a few cm above the soil line.

Phytophthora is never isolated from plants suffering from decline. The fungus *Paecilomyces buxi*, formerly known as *Verticillum buxi*, is rather consistently isolated. Other fungi, including isolates of *Fusarium*, *Phoma*, *Rhizoctonia* and *Pythium* and nematodes of the genera *Pratylenchus, Helicotylenchus, Tylenchus, Xiphenema, Meloidogyne* and *Paratylenchus* have been associated with declining English boxwood plants. Species of *Pratylenchus* and *Helicotylenchus* were reported to be the most commonly associated nematodes. The disease can be induced in the greenhouse by artificial inoculation with the fungus *P. buxi* alone.

When dying English boxwood plants have been replaced with healthy English boxwood, the replants have died within 2 to 3 years. American, Japanese and Korean boxwoods are unaffected and may be used to replace English boxwood.

Although some retardation of symptom expression has been obtained with fungicide drenches and sprays, no satisfactory chemical control has been developed. By the time a declining landscape plant has been identified from foliage symptoms, the root system is so far advanced in decay as to be impossible to save; hence therapeutic fungicide treatments are not likely to be effective in any case. At present, the only recourse is to replace diseased plants with some other shrub.

Nematodes on Boxwood

Boxwoods have been recognized as hosts of nematodes since nematodes have been known as plant pathogens. Several genera of nematodes have been identified from boxwood in the Southeast. Meadow or lesion nematodes (*Pratylenchus* spp.) have been seen as the most destructive parasites of American and English boxwood and root knot nematodes are the most destructive on Japanese boxwood (*B. microphylla japonica*). Nematodes which also may be of importance in damaging American and English boxwood include spiral, ring and stubby root, as well as other species of minor importance. Most reports of nematodes on American and English boxwood have come from Maryland, Virginia and North Carolina. Root knot nematode is more damaging to Japanese boxwood in the more southern part of the range.

Nematode feeding on boxwood results in chlorosis and bronzing of the foliage, reduction in leaf size and eventual defoliation (Color Plate VI, 4). Lesion nematodes produce root lesions which girdle the small roots and cause sloughing of the cortex below the lesion. Proliferation of new roots above the necrotic roots results in a dense mass of roots near the soil surface. Such roots are subject to easy desiccation during periods of drought. Growth is retarded and the plants remain stunted and gradually decline. Root-knot nematodes cause typical galling of the roots with ultimate root decay and stunting and decline of the host. Damage to boxwood from nematodes is difficult to assess since the principal form it takes is slow decline. The loss is sustained largely in landscape plantings. Nematode damage is probably a contributing factor in decline of landscape plantings which results from interaction of nematodes, fungi, winter injury and other stress-inducing environmental factors difficult to define. Commercial production of boxwood should be in soil free of damaging nematodes or in soil treated with a fumigant before planting.

Volutella Blight

Volutella buxi is a fungus which produces conidia in easily detected pink masses on the surface of twigs or leaves of boxwood. These characteristic spore masses are salmon to pink in color and are produced in great abundance on dead and dying tissue, especially under conditions of high humidity. The fungus *Paecilomyces buxi* may also produce superficially similar masses of spores on twigs but they are snow white rather than pink. Microscopically the sporodochia resemble a pin cushion in which slender orange spines (setae), project like needles from the surface of the cushion. The spores of *P. buxi* are not produced in cushions and no setae are formed, helping to distinguish these two fungi from each other.

Symptoms of Volutella twig blight include discoloration and death of twigs, usually restricted to the soft tissue of the current year's growth. Discoloration and necrosis may, in unusual cases, extend well down into woody tissue of the previous year's growth and produce a stem canker. Where colonization by the fungus is extensive, the discoloration below the bark may be discontinuous as in the case of colonization by *P. buxi*. Twigs or leaves kept in plastic bags in cold storage for long periods of time almost invariably develop colonies of *Volutella* at the surface, therefore, diagnosis should be made promptly if damage from other causes is to be distinguished from blight actually caused by *Volutella*. Twig blight occurs among all types of boxwood, perhaps more commonly in American boxwood. The only control that can be recommended is pruning of the diseased tissue below the affected stems. Frequent rains or light syringing of the foliage has been observed to predispose plants to Volutella leaf and twig blight. Wounds on the twigs caused by heavy pruning may become infected by Volutella.

Macrophoma Leaf Spot

The so called leaf spot caused by *Macrophoma candollei* is more properly a leaf blight in which the entire leaf turns yellow. The fruiting bodies (pycnidia) of the fungus appear as elevated black dots on the surface of the affected leaves. The rather large hyaline, single-celled ellipsoidal conidia are produced in great numbers in the pycnidia and ooze out when placed in water.

The fungus frequently colonizes dead or senescent leaves on injured stems or on plants damaged from other causes. It is probably a secondary invader or weak parasite. Root damage or stem injury should be suspected when Macrophoma pycnidia are observed on leaves of boxwood. Remove dead leaves for cosmetic purposes. Fungicides are not recommended for control.

Miscellaneous Disorders

American and English boxwood sometimes have foliage problems of unknown cause. A rather common disorder is characterized by the bright yellow marginal chlorosis of the leaves, especially in clusters of leaves on terminal branches. The result is a green part of the leaf surrounding the midrib and a distinct yellow border. The entire leaf may become chlorotic. Involvement of the foliage of a single plant may be minimal or extensive. A single plant is often the only plant affected in a large planting. The cause is unknown and plants usually grow out of the condition.

A more rarely observed disorder of both English and American boxwood foliage has been seen in which the young leaves are slightly chlorotic and characterized by irregular brownish blotches on the upper surface. The twigs tend to be bunchy and the edges of the abnormally small terminal leaves tend to curl down and there is some twisting of these small leaves. Some leaves turn purplish black to olive in color. The twigs are not involved. The cause is unknown. New leaves and stems that are killed by a late spring or early fall freeze turn very light tan or white.

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Camellia Diseases

Luther W. Baxter, Jr.

Camellia Flower or Petal Blight

Camellia flower or petal blight affects only the open flowers of camellias. It does not affect leaves, stems, roots, non-opened buds or fruit (seed pods). The cause of the disease is a fungus, *Sclerotinia camelliae*, which affects only camellias. It does affect several species of camellia but the fungus is usually active from January 1 through April, coinciding with the normal flowering period of *C. japonica*. Since *C. sasanqua* blooms in the fall, it escapes infection. Buds of *C. japonica*, which are gibbed and thus bloom in the fall, also escape infection.

Most *Camellia japonica* cultivars bloom naturally from January 1 to April 1, depending on location. Along the Gulf Coast they might bloom even earlier than January 1, while in the Piedmont section of the Carolinas they bloom later. Regardless of the time when camellias bloom at their peak (March at Clemson), the fungus *S. camelliae* is active and its growth and spore production is synchronized with this peak blooming period. Activity for the fungus begins with the germination of the fungal survival structure (the sclerotium) which is a black, hard body that generally takes on the shape of the lower part of the old camellia petals (Fig. 21). During germination, a structure called a stipe, grows upward until it comes in contact with light at which time the tip expands to form a saucer-shaped structure called an apothecium (Color Plate II, 2). The apothecia are about the color of pine straw and they are usually about one-fourth to one-half inch in diameter. Regardless of the size of the apothecium, (ium, singular—ia, plural) mature ascospores are produced in the upper surface of this structure and are forcefully ejected into the air currents which distribute them downwind at random. Millions of ascospores are released periodically from these apothecia over a period of several days. There may be from one to a dozen apothecia that arise as the old ones are expended. The ascospores are microscopic in size and are vulnerable to various environmental effects which usually restrict their spread to a few hundred yards, but they can be viable up to a mile. The pathogen *can* spread from camellia to camellia only by wind-borne ascospores released from apothecia.

The fungus responsible for azalea petal blight, *Ovulinia azaleae* is a close relative of the camellia flower blight organism. However, the azalea petal blight fungus produces thousands of asexual spores (non-sexual spores called conidia) which are produced on the diseased flowers and can be spread to other azalea flowers. It is this mass of asexual spores which, during favorable environmental conditions (warm, cloudy, moist weather) cause the almost complete, simultaneous collapse of all the flowers on a particular azalea plant.

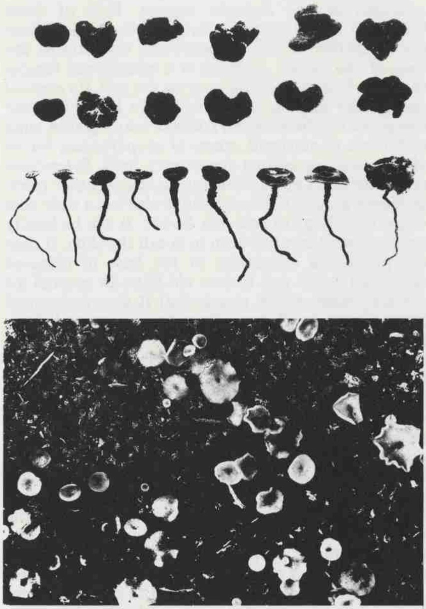

Figure 21. Camellia flower blight. A) Top two rows show sclerotia formed in blighted flowers and bottom row shows apothecial (fungus spore producing structure) that grows from sclerotica. B) Numerous apothecia under camellia bush (NCSU).

Two other fungi need to be mentioned, *Sclerotinia sclerotiorum* and *Botrytis cinerea*. Both of these fungi can attack camellia flowers but neither cause the formation of the characteristic sclerotia at the base of the flower. *Botrytis* is a widespread fungus that survives as sclerotia in or on the soil. Its asexual spores, the conidia, are wind-borne and infect the leaves, stems, flowers and fruits of many plants, such as flowers of marigold, stems of snapdragons, leaves of dogwood, as well as strawberry fruit. *Sclerotinia sclerotiorum* is also active during cool weather (such as March around Clemson) and it also has a wide host range including the camellia flower. It can be locally serious, but it is rarely seen in South Carolina. It does not produce a sclerotium at the base of diseased camellia flowers and it does not have an asexual (or conidial) stage which means that it *does not* spread from flower to flower. *Botrytis* can spread from flower to flower because of the nature of its asexual spores. *Ovulinia* from azalea petals does not affect camellia flowers, and the camellia flower blight pathogen (*S. camelliae*) does not affect other plants.

When ascospores of *S. camelliae* are blown to petals of open camellia flowers, they typically germinate and the developing vegetative growth from the fungus penetrates the flower and then grows throughout the petal tissue. The affected tissue turns light brown in color, particularly noticeable on the white and pink cultivars (Color Plate VII, 3). The disease is equally damaging on the red camellia cultivars but it is not as noticeable on these flowers.

Brown spots are formed within 1 to 2 days after infection when temperatures are in the 60s. When the temperature is in either the 40s or 50s the rate of fungal growth in infected camellia petals is greatly reduced so that it may take 3 to 5 days for symptom expression to develop. After a few days, the lower part of the flower is invaded and a ring of mousy gray fungal tissue forms where the flower base was attached to the plant (Color Plate II, 1). This represents an important diagnostic feature. After 2 to 3 weeks have elapsed, the base of the affected flowers begins to harden and develops into a sclerotium which either will germinate the following year or remain dormant for several years and then germinate.

The disease affects all cultivars of winter and spring flowering camellias. If one has difficulty diagnosing the problem, then place several flowers suspected of having flower blight in a plastic bag (*without additional moisture*), close the bag and leave it for a couple of weeks. The presence of a hard dark mass at the base of the flower will confirm the problem as being flower blight caused by *Sclerotinia camelliae*. This disease can be distinguished from frost injury and mechanical injury by the brown color of diseased tissue compared with a whitish to light tan color resulting from frost injury. Severe freeze injury causes about the same color as flower blighted tissue, but all flowers in an area are affected by a freeze whereas some flowers almost always escape flower blight infection.

Certain cultivars, such as Betty Sheffield (all types) spot severely with water which looks very much like the beginning stages of petal blight, but these lesions from water spotting do not continue to enlarge and no sclerotia form. However, this cultivar can be affected by petal blight so observe the flower carefully.

No control program for this camellia disease is 100 percent effective. If the individual flower is infected by an ascospore, it has the same effect as if it were infected by 100 spores.

The best program for control is to keep the fungus out of the nursery. However, ascospores can be blown in from a neighbor's yard if they have this camellia disease. Unless two nurseries are close to each other (within a mile) there is no danger of wind-borne ascospores being blown from nursery to nursery.

Picking up and destroying the old flowers is still the best control program for flower blight. On plants too small to sell, all flower buds could be removed at one time before they begin to open. However, this program needs to be very thoroughly done, or else a few apothecia can discharge enough ascospores to infect flowers over the entire nursery. Removal of old flowers from container-grown camellia plants is adequate but if a diseased flower falls into a nearby container of other plant species, such as nandina, ligustrum or azalea, then the sclerotia can be taken into a camellia garden unexpectedly. Keep all weeds and other plants that furnish any ground cover out from under camellia plants.

Occasionally ground covers are recommended for growth under camellia plants, such as ajuga, periwinkle, various ivies, Hypericum and many other ground cover plants. However, when infected flowers fall to the ground, these plants act as a moist chamber that enhances sclerotial development. If you have camellia flower blight in your nursery, use pine straw as a mulch. Prune the lower branches so air can circulate freely beneath the plants. This practice permits easy pick-up of the flowers. Otherwise, the flowers will dry up quickly so that sclerotia may not be produced.

Another control procedure is to use chemicals that prevent the completion of the fungal life cycle. These chemicals are applied to the ground surface to prevent the development of the apothecia and the ascospores. These chemicals affect only the apothecium but have no effect on the survival of dormant sclerotia. Sclerotia are resistant to chemicals and also to weather factors such as rain, drought, heat and cold and alternate wetting and drying. Several chemicals have been tested for the control of apothecia by applying them to the soil surface. Two points should be remembered when using any of these materials: (1) a single apothecium can produce millions of spores and so the amount of control achieved by these materials is governed entirely by the thoroughness and effectiveness of the application; and (2) the use of successive applications results in better control because of the good probability of applying the chemicals on spaces missed during the first application. Also, it doesn't matter how well you do the job if your neighbors do not also control the pathogen.

Another approach to flower blight control is to cover the soil with some material such as black plastic which physically deters the development of the apothecia (and thus the ascospores) because light is needed to induce apothecial development. This approach is useful for small areas, particularly greenhouses. The material must cover all areas where the old diseased flowers may have landed.

Still another approach to camellia petal blight control is to spray the flowers and provide some protection during the flowering period. This requires numerous, frequent sprays during the winter and spring and is probably not economical.

Sanitation, exclusion and fall gibbing represent the best control methods yet available. There is a great need for a systemic fungicide that can be applied either to the ground or to the foliage which will control this problem for camellia growers.

Dieback

Camellias thrive in the southeastern part of the United States with the exception of Kentucky, parts of the states of Virginia, North Carolina and Tennessee. Dieback has been known for many years among growers and nurserymen and may be either infectious or non-infectious.

Actually, infectious dieback is a poor name for the disease since the stem does not progressively die back but rather a sudden death occurs of an entire branch or of the entire plant. Close observation reveals a canker (Fig. 22 and 23) at a certain point on the stem which has developed from infection that most probably occurred through a leaf scar (Fig. 25). These cankers develop progressively and infection, leading to a canker, may kill small stems the first year, while large branches, or entire plants may live more than 10 years after the initial infection has occurred before they are killed. Cankers are usually sunken because the dead cells, killed by the fungus, are surrounded by living cells which continue to grow (Color Plate VII, 9). Cankers are much harder to see on cultivars that do not form a ridge of tissue around the canker. Eventually, most of the cells in the canker area of the stem are killed limiting the amount of water which can be conducted through the canker area and thus, under stress, the top dies from lack of water. In this sense it is not a typical dieback as is Phomopsis stem dieback of azalea. The canker could be considered more like girdling a plant.

Infectious camellia dieback is caused by the fungus, *Glomerella cingulata* (asexual state *Gloeosporium* or *Colletotrichum*). The asexual spores are produced on the cankers in the spring of the year when the temperature ranges from about 59° to 77° F (15° to 25°C) and are spread by splashing rains and insects, particularly ants and flies. When they walk on the moist spore masses, they pick up some of them on their feet and then either crawl or fly to other locations laden with the fungus spores. When they land or crawl on newly created wounds, they leave a few spores of the fungus which are adequate to infect the wounded plant.

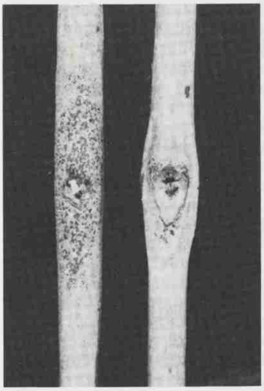

Figure 22. Two branches affected with dieback. Left, no canker development but much sporulation has occurred. This plant has no real resistance. Right, a well-formed canker with little sporulation, denoting some resistance to dieback. Both natural infection (Baxter, Clemson Univ.).

Figure 23. A well-defined canker originating at a leaf scar at a node. Note the dead branch in the center which was artificially inoculated by pulling off a leaf and infecting the plant through the leaf scar (Baxter, Clemson Univ.).

With the exception of tea (*Camellia sinensis*), all camellia species tested are susceptible to dieback. Within the species *C. japonica*, there are varying degrees of resistance to infection. For example, the cultivars Professor Sargent, Governor Mouton and Cho-Cho-San are immune to highly resistant, while the cultivars Guilio Nuccio, Ville de Nantes, Tiffany and Mathotiana are very susceptible to dieback. Several thousand seedlings and many cultivars of *C. sasanqua* have been tested over a period of several years and, thus far, none has been found to be resistant, although there are apparently small differences in susceptibility to this fungus. The symptoms on the stem of a *C. sasanqua* plant infected in the spring (May) could develop into a large canker by fall (September or October) or, the canker could girdle and kill the stem. Rarely does the individual canker become longer than about 50 millimeters (2 inches). When the bark is stripped away from the canker area, exposing the wood, there is chocolate brown dead tissue. The fungus can be recovered from any portion of the dark tissue, (but not from the white tissue which is apparently healthy), when appropriate laboratory techniques are used. Under natural conditions this fungal pathogen does not affect any other known hosts outside of the genus, *Camellia*.

Natural infection by the fungal spores normally occurs through wounds on the plant stem. Lawn mower or pruning wounds provide excellent avenues of entrance but the most natural is infection through leaf scar wounds. When the leaf falls, the remaining scar is subject to infection for 1 day. This may not seem very long, but the most likely time for an old leaf to fall off is generally during spring rain, after the old yellow leaves have fallen. The spores of the fungus are splashed by the same rain at random, some landing on newly formed leaf scars. From impact to infection a leaf scar must be wet only 8 to 16 hours (that is, penetration of the wood by the germinating spore) depending on the temperature.

Another means of infection is during grafting (Fig. 24). During this process, the wood (both scion and understock) is wounded and kept in a very humid environment which facilitates healing of the scion and understock. However, both wounding and high humidity favor disease development. The fungus can grow at temperatures as low as 50°F or as high as 86°F (10°C and 30°C respectively) but at both high and low temperatures the fungal growth is slower. This means that, at these temperatures, the infection period by the fungus on the host is longer than occurs at moderate temperatures, such as 59° to 77°F (15° to 25°C respectively).

The fungus can also infect cuttings, since wounds are created by cutting the stem and by pulling off the lower leaves. Except on resistant cultivars, such as 'Governor Mouton', infection is usually accomplished very easily when the fungus is applied to any leaf scar. (Fig. 25).

Figure 24. Death of a *C. japonica* scion grafted onto *C. sasanqua* understock. Infection occurred through the cut stem of the understock (Baxter, Clemson Univ.).

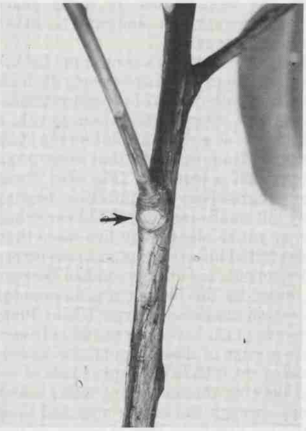

Figure 25. Leaf scar left when old yellow leaf came off. It is at the base of the stem that goes off to the left. This scar is vulnerable to infection for about 24 hours (Baxter, Clemson Univ.).

A point of interest about this fungus is that 8 percent of apparently healthy buds harbored a virulent strain of this pathogen even after the buds had been surface sterilized in clorox.

Here are some steps for control:

1. Grow *C. sasanqua* or *C. oleifera* understock from seed rather than from cuttings. Isolate the seedlings from diseased plants.
2. When collecting scions or cuttings, collect them from healthy plants.
3. Soak scions (or cuttings) in an appropriate fungicidal suspension for 30 minutes just prior to sticking.
4. Stick cuttings in sterilized sand away from any possibly diseased plants.
5. Plant the cuttings or grafts in partial shade (pine is excellent) and keep them properly pruned so that air can circulate freely.
6. When beginning with camellias avoid highly susceptible cultivars such as 'Ville de Nantes' and 'Tiffany'. Instead, use highly resistant cultivars such as 'Governor Mouton' and 'Professor Sargent.' All *C. sasanqua* cultivars thus far tested are susceptible, but 'Mine-no-Yuki', 'Setsugekka', 'Daydream,' 'Apple Blossom' and 'Maiden's Blush' appear to have some resistance.
7. Do not plant newly acquired healthy cultivars near diseased plants.
8. When dieback and/or canker are seen on branches or the main stem, cut out all of the affected tissue (all dark wood) and destroy it.
9. Spray camellia plants with an appropriate fungicidal suspension just after pruning or after cuttings and scions are removed.
10. Spray camellia plants with an appropriate fungicidal suspension during the period of maximum leaf fall. The leaf scars provide a natural wound through which infection occurs.
11. Control insects such as ants with insecticides because they carry the spores from cankers on the stems to the top parts and can carry them directly to the leaf scars.
12. Avoid over-crowding and over-fertilization, particularly with high nitrogen fertilizers.
13. Do not use overhead irrigation during April, May and June on greenhouse grown plants or during May or June on outdoor plants. Overhead irrigation provides a method of spreading the spores to leaf scars where the leaves have recently fallen.

By following these practices, one can reduce dieback and canker, but it is doubtful one can ever completely eradicate this problem on camellias in the South.

Leaf Gall

Camellia leaf gall is a fungus disease that has but one life cycle per year which occurs during late April or May (perhaps slightly earlier in the southern states). Leaf gall, caused by the fungus *Exobasidium camelliae*, affects *Camellia japonica*. *E. camelliae* var. *gracilis* affects *C. sasanqua* and occasionally *C. oleifera* (not to be confused with the *C. sasanqua* cultivar, Narumigata, which is often called "oleifera"), and some hybrids such as Valley Knudson (*C. saluenensis* x *C. reticulata* 'Buddha'). The form of *Exobasidium* that affects *C. japonica* will not affect *C. sasanqua* and vice versa. Also, *Exobasidium vaccinii* that causes leaf gall of azaleas and rhododendron will not attack camellias. Other species of *Exobasidium* also cause leaf and shoot galls on other ornamental and wild plants.

The very distinctive symptoms are characterized by thick, fleshy leaves (Fig. 26). Most leaves in a developing vegetative bud are affected, but occasionally only one or two leaves, or parts of a few leaves, are affected (Color Plate II, 5). The optimum time for symptom expression is May, although the time may vary from location to location. Galls on the affected buds develop at about the time of normal new vegetative growth in the spring. As the galls mature, the lower side of the leaf (lower epidermis) breaks away, exposing the white mass of spores on the surface which are then either wind blown or spread by splashing water. The disease may be very alarming to the grower, but it rarely is damaging except from an aesthetic point of view.

Control involves one of two approaches. First, physical control involves removing and destroying all the galls as soon as detectable. It is difficult to find *all* the galls, but this is absolutely necessary in order to control this disease. (One other problem, your neighbors must also cooperate since the spores can be blown in from outside your nursery.) Secondly, chemical control involves the use of fungicides during the spore production period. Our knowledge about control of this disease by chemical means is not satisfactory, but usually spraying with a fungicide will help. About 3 to 4 sprays applied during May and June should help to control the disease.

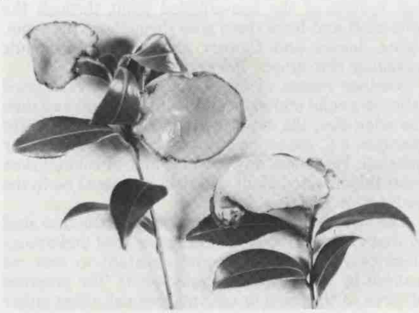

Figure 26. Leaf gall on *Camellia sasanqua* (R. C. Lambe, VPI & SU).

Variegation

It is common knowledge that camellias have either solid-colored or variegated leaves. Variegated leaves are normally green with yellow mottling or splotching (Color Plate II, 3). More spectacular than variegation of the leaves is variegation of red and pink flowers (Color Plate II, 4). Many cultivars are sold as either solid colored flowers or as variegated such as Adolphe Audusson and variegated, Burgundy Queen and variegated, Carter's Sunburst Pink and variegated, Diamond Head and variegated or Don-Mac and variegated. There are two types of variegated flowers. They are infectious (virus-induced) and non-infectious (genetic). In the virus-induced, infectious type, the variegation is irregular and may be slight to severe. Virus destroys color so there is no flower variegation. In white-flowered cultivars, it is impossible to destroy color because white represents the absence of color and thus there is no flower color to destroy. Most virus diseases of plants are harmful but the severity varies from virus to virus as well as the effect of a given virus from host to host.

The virus that causes camellia variegation affects *Camellia japonica, C. sasanqua, C. oleifera, C. reticulata, C. hiemalis, C. sinensis* and camellia hybrids. There are probably many more *Camellia* species susceptible to infection by the virus. Virus of camellias in this country is spread by man during vegetative propagation. There seems to be one exception to the above which is as follows: a solid colored camellia growing alongside a variegated one will sometimes make a root graft and under these conditions the solid one becomes variegated. The virus responsible for this type of variegation (irregular mottling and chlorotic splotching) moves throughout the root system of the affected plant and enters the root system of the non-affected plant through the root graft and from there goes throughout the roots, stems, leaves and flowers and becomes systemic (meaning throughout the system).

Another means of spread is to use a variegated scion on a solid understock. If a union occurs and then the scion dies, the new growth of the understock (for example, a *C. sasanqua* cultivar) will be systemically infected. Therefore, any cuttings and/or scions taken from this infected plant will then be passed on to the cuttings or graft.

The virus is not transmitted through the true seed so that *C. sasanqua* seedlings make good understock since they are virus-free and resistant to root rot (caused by *Phytophthora cinnamomi*). The presence of virus in the scion or cutting does not affect either its grafting or rooting capabilities. Once a plant is systemically affected by a virus it usually remains infected for the remainder of its life although the virus is not equally distributed throughout the plant and symptom expression is variable.

There are several strains of the virus. Some may variegate the leaves but show very little symptom expression on the flowers, while others may show symptoms beautifully on the flower but very little if any symptoms on the leaves. Four virus strains have been reported and there may be others. The symptoms may be so severe that the leaves sunburn and then drop off. If such is the case, some of the defoliated branches may show dieback, but this type of dieback should not be confused with dieback caused by the fungus *Glomerella cingulata,* which kills, not by defoliation, but by destruction of conductive tissue.

As mentioned earlier, variegation of camellia flowers can be caused by genetic variability. Some cultivars, such as 'Herme' and 'Lady van Sittart,' have variegated flowers but the difference is in the pattern. The flower color is regular and appears more as regular stripes throughout the flower rather than irregular white blotches mixed with irregular colored parts of the flower. In this case, one flower of a specific cultivar such as 'Herme' (of a specific type) looks very much like all of the other flowers on that particular cultivar.

Virus variegation may be considered harmful if it causes the loss of the original solid colored scion. However, infection of a cultivar such as 'Adolphe Audusson' by the virus, may greatly enhance the beauty of the flower.

Root Rot

Root rot of camellias, particularly *Camellia japonica,* is caused by the fungus *Phytophthora cinnamomi.* This fungus attacks many ornamental plants plus various pine species, particularly short leaf pine, *Pinus echinata,* causing little leaf disease. Several pine species are used as partial shade for growing camellias and this is highly desirable since camellias do better in shade of this type. Thus, the pathogen may already be present in the soil when camellias are planted. It can also be a serious problem in container grown plants.

Camellia plants with Phytophthora root rot show symptoms of general decline, low vigor and lack normal dark green foliar color (Fig. 27). Such plants often show symptoms of twig dieback that can be confused with the dieback or canker disease caused by *Glomerella cingulata.* Plants of any age can be attacked. Infected plants die gradually over several years or may die completely in just a few weeks. When above ground symptoms become apparent, the root system is often completely destroyed.

Since *Camellia sasanqua* is highly resistant to Phytophthora root rot, it has been a standard practice for many years to graft other camellia species onto *Camellia sasanqua* seedling root stocks. Due to the high cost of grafting plants, many nurseries at present produce camellias from cuttings. For more information see the general section on Phytophthora root rot and its control.

Figure 27. Phytophthora root rot of *Camellia japonica* Blood of China (Healthy plant on left) (R. C. Lambe, VPI & SU).

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Crab Apple Diseases

John Hartman

Fire Blight

Fire blight is a serious and damaging disease of flowering crab apples. Its distribution is almost as general as that of scab, but its yearly occurrence is much more sporadic. The severity of infection depends on a source of inoculum, susceptible trees and warm moist weather during the bloom period when much of the infection takes place. Areas of Colorado and North Dakota may have severe fire blight infections almost every year, while in some areas of the southeastern states severe infections may not be seen for several years. When severe infection does occur it may be devastating, as several branches or whole trees may be killed outright.

In addition to crab apples, apples, pears and quinces, the fire blight organism attacks a number of other ornamental plants including firethorn, cotoneaster, hawthorn, mountain ash, loquat and serviceberry.

Symptoms of infection first appear on blossoms about the time or soon after the time that petals fall. The infected blossoms appear to be water soaked and are killed rapidly, with accompanying shriveling and browning. After the bloom period, wilting and brownish-black dieback may occur on terminal growth, on water sprouts and on shoots at the base of the tree. Dead leaves may remain attached after the twigs die. Affected shoots often curve near the tip, forming a shepherd's crook. Affected blossoms, twigs and branches turn brown as though scorched by fire (Color Plate VIII, 6). Cankers may occur on small or large limbs, trunks or roots and they usually start around the base of a blighted blossom spur or shoot. Cankers are slightly sunken areas of dead bark, surrounded by an irregular crack or wound callus-layer.

The cause of fire blight is *Erwinia amylovora*. In the tissue of the apple, the bacteria grow in intercellular spaces and produce gelatinous strands consisting of slime and bacterial cells. Under favorable conditions, the bacteria reproduce rapidly and can be seen as viscous clear, amber or creamy white ooze on the surface of infected tissues.

The fire blight bacterium survives from year to year in the margin of cankers. As soon as the sap flows in the spring, the bacteria resume activity. They increase rapidly in number and are pushed to the surface where they form droplets of bacterial ooze.

Splashing rain may dislodge the droplets and spread bacteria to opening blossoms. Insects, particularly bees, are attracted by the sugar contained in the ooze and become contaminated with the bacteria. As insects move about in search of nectar, the bacteria are transported to open blossoms of crab apple and other susceptible species where, in humid weather, they enter the nectaries of the blossom.

Once the fire blight bacteria become established in a blossom, they migrate through the stem into older blossoms of the cluster and may be carried to terminal shoots and water sprouts. On susceptible crab apples, the bacteria may move from the terminals and blossoms to the larger limbs where cankers are formed. By July or August the margins of twig and limb cankers become distinct, and barriers of wound callus are formed by the tree at the canker margin.

Conditions favorable for fire blight infection include open blossoms or succulent new growth, temperatures above 65°F, long frost-free periods before bloom plus rainfall or relative humidity above 60 percent. The disease is most likely to be severe in areas where fire blight was present in the preceding year.

The following suggestions will aid in control of fire blight in flowering crab apple.

1) Removal of sources of overwintering bacteria—Cankers on old neglected pear and apple trees are important sources for overwintering fire blight bacteria. Such trees should be removed and destroyed in the immediate vicinity of nurseries and other areas where crab apples are grown.

2) Spraying—Blossom sprays which help to protect the open flowers from infection are an important measure in fire blight control. Apply a recommended antibiotic at 4 to 5 day intervals during bloom. To prevent outbreaks of fire blight from occurring on new terminals and suckers during rainy, windy, humid periods following bloom, apply sprays of the antibiotic at 7-day intervals.

3) Removal of early infections following bloom—About 2 weeks after petals fall, all trees should be inspected for infected shoots and branches. All infected branches should be broken off at once and destroyed. Several additional inspections should be made at weekly intervals.

4) Sucker removal—On all varieties of susceptible crab apples, succulent suckers or water sprouts should be removed as soon as blossom blight starts to appear. Suckers that develop from roots, trunk and scaffold branches are ideal places for the fire blight bacteria to enter limbs and produce cankers. Before the suckers become woody they can be pulled off easily. Blighted suckers must be cut with tools disinfested between cuts with a solution of 1 part household bleach (clorox) and 9 parts water or 70 percent alcohol.

5) Cultural practices—Since new growth is susceptible to fire blight, any system of culture that does not promote excessive succulent growth will aid in reducing the amount of infection. Soil management and nutrition practices should be arranged to prevent sudden increases in supply of nitrogen which would encourage growth late in the season. Heavy applications of nitrogen should not be made, and sources of organically bound nitrogen like barnyard manure should not be used. When tree growth stops early, fire blight lesions are arrested sooner and less direct damage to the tree may result. Nurserymen should maintain the pH of the soil at 5.5 to 6.5.

6) Resistant varieties—Fire blight resistant types are listed in the chart at the end of this section.

Scab

Scab causes severe losses of the commercial apple crop and it can also be a serious problem causing spotting of the leaves, premature defoliation and unsightly corky spots on the fruit. A fungal disease, scab is found whenever crab apples are grown in humid climates but it is not usually a problem in the semi-arid region of the Rocky Mountain states.

The appearance of dull, smoky areas on the new leaves is the first evidence of the disease. These areas soon become olive colored and velvety (Color Plate VII, 5). As the infected areas become older, they assume a definite outline as olive green or brown circular spots. Often the leaf tissue underneath the spot is raised or puckered. As more foliage develops, it is equally susceptible and the characteristic spots may appear on leaves anywhere on the tree. When early, severe foliage infection takes place, the leaves yellow and the tree may lose almost all its leaves by midsummer. Although this premature leaf fall gives an unsightly look to the trees, it does not seem to have any permanent ill effect on the growth of landscape trees. However, early defoliation of nursery trees will cause slow growth and result in fewer saleable trees in subsequent years.

Fruit may become infected at any time in its development. Typical fruit lesions are distinct, almost circular, rough-surfaced, olive-green spots which turn brown to black. This discoloration lowers the ornamental value of the fruit.

The scab fungus (*Venturia inaequalis*) overwinters in old, infected fallen leaves. Fruiting bodies are produced within dead leaf tissue. In the spring these bodies produce ascospores, which are discharged into the air currents and are carried to developing apple buds and foliage. Fruiting bodies must be wet for spore discharge to take place. If spores land on crab apple foliage or fruit and if the tissue surface is wet and remains wet for several hours, the spores will germinate and send sprouts or germ tubes into the tissue. After 8 to 18 days depending on temperature (development occurs most rapidly at warm temperatures) a visible spot is produced. On the surface of the infected area conidia are produced. These spores are easily dislodged and are splashed by rain to new leaf and fruit surfaces within the tree. In this manner, several secondary cycles of infection may occur in the course of a growing season.

Another source of secondary infection may be from spores produced on scab lesions found on the twig growth of the previous year of severely infected susceptible crab apples such as *Malus* cv. Almey, *M.* cv. Hopa and *M.* X *purpurea* cv. Eleyi. In addition, these scab lesions on the twigs of susceptible trees serve as an important means of introducing the scab fungus from the nursery to plantings where the disease does not already exist.

Scab infections may be prevented by 5 to 8 applications of fungicides at 7 to 10 day intervals starting as soon as leaf growth appears. The critical period for scab control is from the time bud growth begins until about the middle of June. The object is to provide a protective coating of fungicidal chemical that will kill any spores already present on the leaves and fruit and any that may subsequently land on the surface. If scab infection can be prevented until all ascospores are discharged from old leaves, the cycle is broken and little further source of infection remains for the rest of the season. However, if early control is not obtained and if leaf and fruit infection do occur, conidia will be produced on these lesions, and scab will be a constant threat whenever wet weather occurs during that season. Use of scab resistant cultivars is the best means of control. Refer to the chart at the end of this section.

Cedar-Apple Rust

Cedar-apple rust is usually a problem only in areas where large numbers of native cedars or plantings of ornamental junipers are growing in close proximity to the crab apples. This situation could occur almost anywhere in the eastern half of the United States.

The symptoms appear on affected crab apple leaves as yellow to orange areas one-eighth to three-fourths inch in diameter (Color Plate VII, 8). The upper surface of these areas is covered with minute black dots within a reddish circle. Later, on the under leaf surface of the orange spots, many one-eighth inch diameter cup-shaped structures with fringed edges are formed in circular clusters. Severe infection of leaves and twigs may cause early leaf fall and dwarfing of the tree. On the very susceptible Bechtel crab apple, repeated infection may cause death of some branches or of the entire tree.

The cedar-apple rust fungus (*Gymnosporangium juniperi-virginiana*) is a basidiomycete fungus with a complex life cycle that involves production of several kinds of spores and involves infection of two different host plants every year. Spores carried by air currents from infected cedar trees germinate and penetrate directly into the crab apple leaf to cause infection. Spots are formed on the upper surface of infected crab apple leaves in early summer and spores of another type are produced in an ooze that is attractive to insects. Spores of one spot are spread to nearby spots by the insects, thus allowing fungal mating to occur. In late summer, "cluster cups" of the rust fungus are produced on the underside of the infected apple leaves and still another kind of spore is released and carried by air currents to nearby juniper plants. The cycle is completed about 20 months later when galls on infected junipers produce orange colored tendrils with spores capable of infecting crab apple.

Infection of crab apple in the nursery depends on: 1) the amount of rust development on nearby cedar or juniper; 2) wet weather in the spring when apple leaves are being infected; and 3) locations of nearby cedars and junipers in relation to the crab apples, (the greater the distance, depending on terrain and wind direction, the less infection). If cedars and crab apples are separated by more than a mile, very little infection is likely to occur.

Elimination of cedar trees within a mile of the orchard provides almost complete control. However, cedar trees are often on neighboring property or in landscape plantings that cannot be removed, thus remaining a source of spores for infection of the crab apple foliage. Three applications of approved fungicide made at 10-day intervals starting about the time color shows in the blossom buds will provide effective control of the disease. Use of disease resistant crab apples is the most effective method for control of cedar-apple rust. See the chart at the end of this section.

Black Rot

The disease, caused by the fungus *Physolospora obtusa*, is also referred to as "frogeye leaf spot" due to the appearance of leaf lesions. Fruit infections and branch cankers may be present as well. In the southern areas of the United States, fruit and leaf infections are most common, although cankers also occur.

Leaf lesions appear first as small purple specks, then enlarge to about one-eighth to one-fourth inches in diameter. Older lesions have irregular margins and centers with alternating bands of light and dark giving the lesions a "frogeye" appearance. Later, black spore producing structures (pycnidia) may develop on the upper leaf surface, near the lesion centers. Infected fruit develop a brown to black rot with alternating light and dark bands around the calyx (blossom) end of the fruit or in an injury. The rotted tissues remain quite firm. Lesions continue to expand in a concentric manner until the entire fruit is rotted. Pycnidia often develop within mature fruit lesions. Severely diseased fruit may dry up and persist in the trees as mummies. Infected areas on limbs and branches are reddish-brown and slightly sunken. Cankers expand a little each year and eventually become several feet in length. The bark on older areas of the canker can be removed easily whereas recently killed bark clings firmly. Infected branches may be weakened enough to break; sometimes they are killed outright. Cankers found in branches were first weakened by heavy shading, sun scald or winter injury. Pruning cuts and old fire blight cankers are major points of entry. The disease is most often found on older trees; young nursery trees are rarely troubled by the disease.

The black rot fungus overwinters in cankers, on dead wood and in mummied apples. In spring, conidia are formed in pimple-like pycnidia found on the surface of diseased tissues. Perithecia may also be produced in cankers and discharge ascospores in spring.

During rain, conidia ooze from the pycnidia and are spread by splashing raindrops to the fruit, leaves and branches where infection occurs if weather remains favorable. Black rot infection of leaves and fruit commonly develops in cone-shaped areas in the tree beneath black rot mummies or old fire blight cankers. The heaviest discharge of spores comes at the bloom stage of tree development. Once released, conidia remain visible over extended periods. Spore germination is favored by high temperatures and this may explain in part why the disease is much less of a problem in northern areas.

Sanitation or the removal of diseased branches and of mummies is primary to control. Prunings should be removed from the nursery and burned by the early pre-pink stage. Maintaining trees in good vigor by adequate pruning and fertilization helps trees to resist infection. Trees should be sufficiently open to allow spray and sunlight penetration. In areas where the disease is common on leaves, a fungicide spray program is required from early spring through harvest. Resistant cultivars are available and should be grown.

Powdery Mildew

The powdery mildew fungus (*Podosphaera leucotricha*) attacks leaves, terminals, blossoms and fruit. The most common symptoms are twisted, narrow, cupped terminal leaves covered with a white powdery fungus coating (Color Plate VII, 7). Infected terminals have weak buds and may winterkill easily. On susceptible cultivars such as Almey, white powdery patches of the causal fungus may be found on the fruit.

Powdery mildew is usually a problem on crab apples only in locations where the air movement around the trees is poor or where they are growing near an orchard of susceptible apples such as Cortland or Rome. The fungus overwinters as fungal threads within dormant buds. High humidity and temperatures around 70°F provide ideal conditions for development of the disease. The spores will not germinate in free water on the leaves, but rather require high humidity.

As soon as the disease appears, which may be any time after the blossoms open until midsummer, spray 3 times at weekly intervals with an approved fungicide. There are powdery mildew resistant crab apples available and these should be used whenever possible. See the list at the end of this section.

Disease resistant and susceptible flowering crab apples are presented in the following tables. (Information compiled by L. P. Nichols, Pennsylvania State University).

Table 11. Crab Apple Cultivars and Varieties Resistant to Scab, Rust, Fire Blight, Powdery Mildew and "Frogeye Leaf Spot."

Ames White	Milton Baron #2
Autumn Glory	Molton Lava
Centurion	Morden 19-27
Christmas Holly	Mount Arbor Special
Coral Cascade	Professor Sprenger
Cotton Candy	Red Swan
Donald	R. M. J. 102
Gibb's Golden Gage	Robinson (10)
Golden Gem	Tina
Harvest Gold	Weis
Henningi	
hybrid (scab immune clone-GR 700-58)	

Table 12. Crab Apples Having Slight Susceptibility (moderate or high if indicated) to One or More Diseases: Scab (S), Rust (R), Fire Blight (F), Powdery Mildew (PM), "Frogeye Leaf Spot" (L).

Cultivar	Susceptible to
cv. Adams	F, PM
Albright	S, F, L
Arctic Dawn	R (mod), PM
baccata var. *Jackii*	F (high), PM
baccata var. mandschurica	S, PM (mod)
Beverly	F (high)
Bob White	F (mod), PM
Brandywine	S (mod), R
Burgundy	S (mod)
Burton	S
Callaway	R (mod), F, PM
Candied Apple	S (mod)
Coralburst	S
David	F
Dolgo	S, F
Donald Wyman	F (high), PM
Ellen Gerhart	S (high), R, PM
Ellerwangiana	F (mod), PM
floribunda	F (mod), PM, S
Golden Hornet	F (high)
Henry Kohankie	S
hupehensis	F (high)
Indian Magic	S (high), R
Indian Summer	S (mod), F
Inglis	F, R (mod)
Jewelberry	S, F
Kibele	S, F (mod)
Liset	PM (mod), F, S
Makamik	PM (high), F
Mary Potter	S, F, PM (mod)
Ormiston Roy	F (mod), R, S
Profusion	PM (mod), F, S
Red Baron	S (high), F, R
Red Jewel	F, S (mod), R
Red Splendor	S (high), PM, F (mod), R
X robusta Persicifolia	S, F
rocki	PM (mod)
Royalty	S, F (high)
Royal Ruby	S (high), PM
Ruth Ann	S
sargentii	F (mod)
Selkirk	S (high), PM (mod), F
Sentinel	S, F
Shaker's Gold	F (high)
sieboldii Fuji	S, PM (mod)
sieboldii var. *zumi* Calocarpa	F (high), PM, S
Silver Moon	F (high), S (mod), L
Snowdrift	F (high), S
Spring Snow	S, F (high)
tschonoskii	F (high), S
White Angel	R (high), F (mod), S
White Candle	S (high), F (mod), PM
Winter Gold	S (high), F (mod), PM

Table 13. The Following Crab Apple Cultivars Should Not Be Used in the Nursery Trade Because of Extreme Disease Susceptibility.

Alamata	*lancifolia*
Almey	Leslie
Amisk	X *micromalus*
angustifolia	Neville Copeman
X *arnoldiana*	Oakes
Arrow	Oekonomierat Echtermeyer
brevipes	Patricia
Brier	Pink Perfection
coronaria	*platycarpa*
coronaria	*prunifolia rinki*
Charlottae	Purple Wave
Cowichan	X *Purpurea* Eleyi
Crimson Brilliant	X *scheideckeri*
E. H. Wilson	Scugog
Flame	Snowcloud
florentina	Strathmore
Frau Luise Dittmann	Vanguard
Geneva	Wabiskaw
glaucescens	Wynema
Goldfinch	Young America
Henrietta Crosby	
Hopa	
ioensis	
ioensis	
Plena	
Irene	
Jay Darling	
Jubilee	

Crapemyrtle Diseases

James M. McGuire

Powdery Mildew

Powdery mildew, caused by *Erysiphe lagerstroemiae*, is the most important disease of crapemyrtle. It is usually most serious in spring and early summer and in the fall on outside plants, and it occurs year around on greenhouse and lathhouse potted plants. It can be especially severe in the greenhouse on plants grown for forcing.

The white mycelium and spores of the fungus coat leaves, shoots and inflorescences causing stunting and distortion, leaf curling and failure of flower buds to open properly. The entire plant may be affected.

There seems to be some variation in severity on different cultivars, especially the dwarf types. The cultivars, 'Catawba', 'Cherokee', 'Conestoga', 'Potomac', 'Powhatan' and 'Seminole', released since 1962 by the U. S. National Arboretum, are mildew tolerant. The cultivars 'Muskogee' and 'Natchez' released by the National Arboretum in 1980 are reported to be highly resistant to powdery mildew. In greenhouse and nursery, general sanitation and good air circulation is important in powdery mildew control on susceptible cultivars. For more information, see the general section on powdery mildew. Fungicides effective against powdery mildew disease may be necessary occasionally. Make 2 to 3 applications 10 to 14 days apart.

Sooty Mold

Sooty mold, caused by *Capnodium sp.*, is a dark brown or black growth of fungus in blotches or as a complete coating on leaves and stems of plants. It results from a non-parasitic fungus which grows on "honey dew" excretions of insects such as aphids and scale. It is a common problem on crapemyrtle when the crapemyrtle aphid is not controlled.

The unsightly coating of the sooty mold fungus can be easily wiped or washed from the plant parts. It does not damage the shrub or tree unless a heavy coating, which will interfere with the amount of light that reaches the plant, is left unchecked.

Control of sooty mold is accomplished by insecticide applications to control aphids and scale insects. When the insects are controlled, no food supply is available for the sooty mold fungus and it cannot grow.

Leaf Spot

Cercospora leaf spot, caused by *Cercospora lythracearum*, is a common disease of crapemyrtle from midsummer through fall in southern areas during wet or humid conditions. The fungus causes large, dark brown spots to develop on leaves starting at the bottom of the plant and progressing upward. The leaves soon develop large yellow areas around each spot and defoliation occurs. Even one or two spots will cause a leaf to drop. In severe cases, 75 to 80 percent of leaves may be lost from a plant prior to frost. This premature defoliation weakens the plant over a period of years and will reduce its longevity.

Spores of the fungus are produced on the surface of diseased leaves and are spread by wind or rain. Large numbers of spores are produced, and secondary spread to other leaves is rapid.

Dogwood Diseases

R. C. Lambe and R. K. Jones

Diseases may be an important factor in the production of saleable flowering dogwood (*Cornus florida*). Recently several different virus diseases have been reported, but little is known about their impact on the production of dogwood. Historically, fungus diseases of the foliage, twigs, roots and trunks have been considered important. These diseases occur frequently under certain environmental conditions of excess rainfall, water-logged soils and low temperatures. Recently a trunk canker of undetermined cause has assumed an important position in the commercial production of dogwood.

Foliage and Flower Diseases

Ascohyta leaf spot, caused by the fungus *Ascochyta cornicola*, was first reported in 1942 at Biltmore, North Carolina. Leaf spots appear as early as mid-June and are round or slightly irregular areas, ranging in size from 1.5 to 6.5 mm in diameter. Tiny black spore producing pycnidia form on gray to tan spots surrounded by a somewhat prominent brown to red border.

Botrytis blight, due to infection by the fungus *Botrytis cinerea,* appears on bracts as brown patches or large, irregularly shaped lesions with a wrinkled appearance. Succulent leaves and young shoots may also be infected. The disease generally occurs during periods of cool, wet weather. In humid weather, lesions become covered with fuzzy grayish-brown fruiting bodies. Other symptoms include discoloration or fading of bracts, leaf rotting and the occurrence of blemishes on any floral parts, leaves and shoots.

Cercospora leaf spot, caused by the fungus *Cercospora cornicola,* occurs as irregular brown areas without definite borders, 5 to 10 mm in diameter. The disease appears in late summer or fall and often causes defoliation.

The fungus *Colletotrichum gloeosporioides* causes necrotic spots on foliage. Defoliation and dieback of 1-year cuttings and seedlings caused by this pathogen have been observed.

The disease, caused by the fungus *Elsinoe corni* and referred to as blossom blight, blossom spot or spot anthracnose, is the most important foliar (Fig. 28) and flower disease on dogwood. This disease was first reported in 1948 as a disease disfiguring the bracts. The fungus infects bracts, foliage and twigs in developing stages. Petioles, peduncles and fruit clusters may also be infected. Leaf spots are circular, angular or elongate, ordinarily 1 mm in diameter, but sometimes larger. Spots may number over 100 per leaf causing young leaves to be deformed. Dead tissue in the centers of spots becomes pale, yellowish-gray and readily drops out, while surrounding tissue may darken. The disease may reduce leaf size or kill tissue outright. When flower bracts are heavily infected before they fully open, the bracts may never open or remain very deformed. Bract spots are generally circular, 1 to 3 mm in diameter and pale to clear with a reddish border (Color Plate VIII, 3).

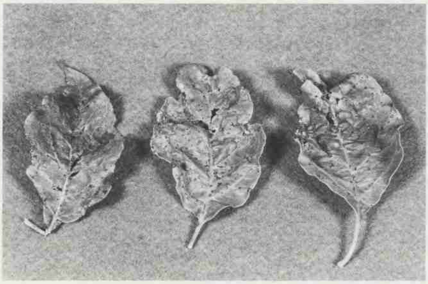

Figure 28. Spot anthracnose of dogwood leaves (R. C. Lambe, VPI & SU).

Disease development is favored by cool temperatures and abundant moisture. These conditions are almost always present in early spring in containerized nursery dogwood trees, watered frequently with overhead irrigation systems. Under such conditions new leaves can be heavily spotted, deformed and plants stunted. There is a frequent variation in disease severity between trees in the same planting. Anthracnose is generally less severe on field grown trees with infrequent or no overhead irrigation. Severely diseased trees are stunted and greatly reduced in quality. Since the fungus overwinters on stems and berries and is carried to landscape plantings on diseased nursery stock, anthracnose should be controlled in the nursery.

Control—Dogwood grown in containers appear to be more susceptible to spot anthracnose than field grown plants. Irrigation should be timed so that the foliage dries rapidly. Fungicides should be so timed as to protect the newly developing leaves in the spring. Improve air circulation by spreading containerized plants further apart. Fewer fungicide applications will be necessary on field grown dogwood. Fungicides used to control anthracnose will also control the other leaf diseases.

Elsinoe floridae, as well as *Ramularia gracilipes* are mentioned as leaf spotting fungi on flowering dogwood. *Phyllosticta cornicola* and *P. globifera* can be easily confused with *Ascochyta cornicola* in early spore stages.

The fungus *Septoria cornicola* is the cause of a leaf spot having small, angular and typically haloed margins. *Septoria floridae,* the cause of Septoria leaf spot, produces small angular leaf spots which are limited by veins. Color is generally uniform but may become light in the center and dark at the borders. Small black pycnidia form late in the summer on the necrotic centers.

Fungi causing twig blights reported on dogwood include *Botryosphaeria dothidea, Corticum stevensii* and *Myxosporium* sp. These are primarily damaging on older plants under stress.

Root Rots

Phytophthora root rot has been observed in field plantings of dogwood. *Phytophthora cactorum* causes crown rot on flowering dogwood. When seedling nursery stock is planted in fields having poor surface- or internal-drainage, *Phytophthora cactorum* has been isolated from diseased roots.

Colonization of roots by *P. cactorum* and *P. cinnamomi* has been shown to occur in wet soils without the presence of above ground disease symptoms. However, a high water table or excessive soil moisture may predispose the plants to infection and also provide favorable conditions for production, liberation and dispersal of zoospores. These high soil moisture conditions are typical, at times, in many areas. Such trees often die after being transplanted to the landscape.

Phytophthora citricola has been reported to occur both as a crown rot and a root rot organism in several nursery crops. Similarly, *P. cactorum* which was previously reported as causing a crown or collar rot, has been shown to cause wilt and root rot in dogwood under conditions of high moisture, and therefore must be considered as a potentially serious pathogen of dogwood.

Select fields that are well drained and avoid those that have heavy subsoil structure. Placement of drain tiles may be necessary to remove excess water. Dogwood should be grown in well-drained, porous media in containers.

Trunk Diseases

Botryosphaeria dothidea (*ribis*) has been reported as a twig blight and also a canker fungus, occasionally causing dieback, killing branches and whole trees in the northeastern United States. *Nectria galligena* causes the development of zonate cankers with conspicuous bark-free callus tissue (Fig. 29). This area folds and tiny red, balloon-shaped perithecium develop around the edges in wet weather.

Figure 29. Dogwood canker caused by the fungus *Nectria galligena* (R. K. Jones, NCSU).

The bacteria, *Agrobacterium tumefaciens*, occasionally causes stem and crown galls. *Pythium* spp. have been the cause of seedling root rot or damping-off in wet, poorly-drained soils.

A previously undescribed canker has been seriously affecting dogwood growing in nurseries in several eastern and southern states and in the landscape for at least the last 13 years. The disease is becoming more common, affecting hundreds of dogwood in various regions with serious economic implications for certain nurseries. Other states reporting a similar canker include Tennessee, Arkansas, North Carolina, South Carolina and Ohio. No causal organism has been identified, although numerous experiments have been conducted and research is continuing.

Cankers are first seen on the main trunk at or above the soil line. On young trees, cankers first appear as a very slight roughening of the bark (Fig. 30). Two kinds of cankers have been observed and with one of these, bark cracking and localized swelling occurs. As the trunk enlarges, the rough areas become deeply fissured. Cankers on the main trunk may then appear at different points on the trunk above the ground (Color Plate VI, 1). No fungi or other organisms are visible on the surface of the trunk. Boring insects sometimes invade the cankers and cause severe damage during feeding. A second type of canker appears on small trees as a constricted or sunken area resulting in girding and death of the top. Trees with cankers often produce numerous sprouts (Fig. 31).

Figure 30. Dogwood canker (Lambe, VPI & SU).

Figure 31. Development of numerous sprouts on dogwood tree with canker (R. C. Lambe, VPI & SU).

In a group of trees from the same source, we have observed that certain trees will have cankers shortly after they are planted while other trees will be free of cankers and remain so for several years even though they are in close proximity to cankered trees.

When three groups of two-year-old seedlings from three different sources were planted in the same research plot and grown under the same conditions, the incidence of canker varied widely. After two growing seasons, the percentage of cankered trees from the three sources was 0, 5 percent and 50 percent. Numerous cankered trees died during the second growing season in the most seriously affected group. However, in some instances the roots survived and produced sprouts. Some of the trees were still uncankered after 5 years.

In a commercial dogwood nursery, trees of different cultivars grafted on white seedling roots were evaluated for cankers. The pink cultivar Cherokee Chief was much less severely cankered than the white seedlings, whereas cankering was only slightly less in the white cultivar Princess.

One-half of a group of 100 one-year-old seedlings were pruned and the other half left unpruned in order to determine the effect of pruning on canker development. The trees were planted in a field where dogwood had not previously been grown. At the end of the second growing season, the incidence of cankers per tree on pruned trees was 1.01 and unpruned 1.06 per tree.

The source of trees appears to be important. Most dogwood seedlings are purchased free of visible cankers only to become cankered within one or two growing seasons. Fungicides ordinarily applied for leaf spot disease protection have been ineffectual against dogwood canker. This would suggest a pathogen that is unaffected by fungicides applied to outer plant surfaces, if the pathogen is indeed a fungus.

Viruses

In recent years arabis mosaic virus, broad bean wilt virus, cherry leafroll virus, cucumber mosaic virus, tobacco ring spot virus and tomato ring spot virus have been reported in landscape dogwood trees.

Leaf symptoms include mosaic patterns, chlorotic spots or ring spots, yellowed tips and occasionally an oak leaf pattern. Leaf symptoms may disappear during the summer months. Visible leaf symptoms may seldom be apparent with these viruses. Flower bracts may be elongated or twisted. Virus infected trees may be less thrifty, flower sparsely, appear stunted and have twig dieback. It has also been suggested that virus infected dogwood plants are more susceptible to spot anthracnose.

While these virus diseases of dogwood have not been reported in nursery age trees, it may be that symptoms are not expressed in young vigorously growing trees. A systematic assay of nursery dogwood trees has not been done to determine the possible incidence of symptomless virus infection. How these viruses are spread in dogwood and how frequently they are spread is unknown.

To minimize the occurrence and spread of these virus diseases in nursery production of dogwood, however, the following precautions are suggested:

1) Carefully select healthy vigorous trees as seed sources, as several of these viruses are seed transmitted in other host plants. During and soon after the flowering period is the best time to check seed trees for virus symptoms.

2) Dogwoods should be seeded or transplanted into fumigated soil or soil known to be free of the following nematodes—

a) *Xiphinema diversicaudatum*—transmits cherry leafroll and arabis mosaic viruses.

b) *Xiphinema americana*—transmits tobacco and tomato ring spot viruses.

These nematodes and viruses have very wide host ranges in both wild and nursery grown fruit and ornamental plants. These viruses survive in the above mentioned nematodes in the soil in the absence of a susceptible host.

3) Carefully select healthy trees for sources of grafting wood as all of these viruses can be graft transmitted.

4) Do not grow dogwood from cuttings as all of these viruses can be transmitted through cuttings.

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Euonymus Diseases

R. C. Lambe and J. J. McRitchie

Anthracnose

Anthracnose leaf spot of euonymus is caused by the fungus *Colletotrichum gloesporioides*. Under certain environmental conditions, the plants can become severely damaged as to lose most of the leaves and suffer a stem dieback, making the plants unsaleable.

Under container culture several cultivars of *Euonymus japonica* are seriously defoliated by anthracnose (Fig. 32-34). Frequent irrigation and rains create optimum conditions for disease. A disease of similar symptoms called scab (*Elsinoe euonymi-japonica*) has been reported on euonymus, but is not as common in the southern states as anthracnose.

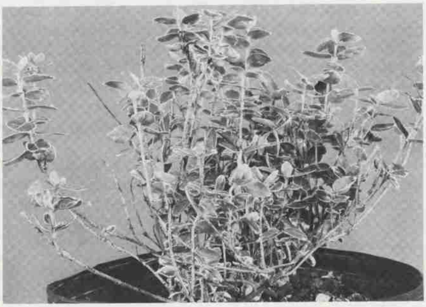

Figure 32. Variegated euonymus showing defoliation, dieback and leaf and stem lesions typical of anthracnose (R. C. Lambe, VPI & SU).

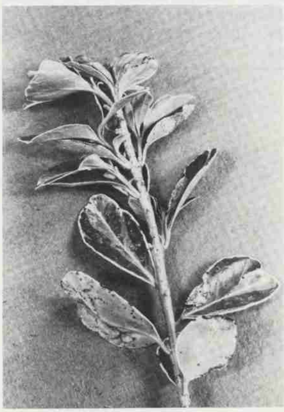

Figure 33. Anthracnose on variegated euonymus (R. C. Lambe, VPI & SU).

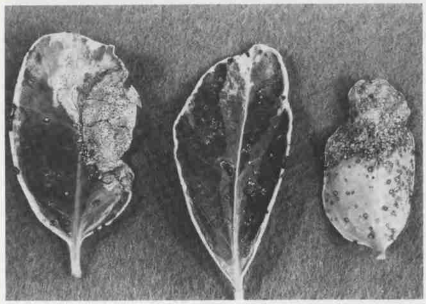

Figure 34. Anthracnose on variegated euonymus (R. C. Lambe, VPI & SU).

The modern use of the term "anthracnose" refers to a plant disease characterized by zonate lesions of foliage, fruit and stems often accompanied by dieback. Anthracnose is also used to describe diseases caused by acervulus-forming fungi of the genera *Colletotrichum* and *Gloeosporium*.

Colletotrichum spp. are capable of overwintering in the previously infected host tissue, most likely as vegetative mycelium, and are dependent on periods of precipitation and high relative humidity for spore dispersal, germination and penetration. These fungi are capable of repeated infections of new tissue throughout the growing season. After a 48-hour waiting period, extensive new anthracnose lesions are evident on both leaves and stems. New lesions are so numerous as to coalesce and many leaves abscise. Acervuli appear as small black dots in the established lesion centers, especially after periods of precipitation and high humidity. In all cases where new infections are detected, relative humidity is greater than 90 percent for at least 24 hours after 48 hours of leaf wetness.

The disease appears on the leaves as distinct circular, dark brown lesions measuring from 0.5 to 3.0 mm in diameter with light tan necrotic centers. The spots occur on the upper and lower leaf surface (Fig. 33-34). During the later stages of infection, a reddish discoloration may be noted in the tissue surrounding the lesion and necrotic lesion centers may drop out and create a "shot-hole appearance." Stem lesions appear as raised, brown, circular to elliptical necrotic spots measuring from 0.5 to 3.0 mm in diameter with a light tan center.

Control of anthracnose is dependent on the particular host plant and prevailing weather conditions. Anthracnose is usually more prevalent during warm wet weather conditions or frequent irrigation in nurseries during summer months; therefore, chemical control measures are most beneficial at such times. It has also been established that anthracnose pathogens overwinter in previously infected foliage, stems and petioles of living plants. When economically feasible, sanitation, such as removing diseased prunings and removal of dead stems, may be of value in the control of anthracnose diseases. Cuttings must be taken from disease free plants.

Crown Gall

Crown gall, caused by the bacterium, *Agrobacterium tumefaciens*, has a very broad host range which involves over 40 families, both herbaceous and woody. Many woody ornamentals are susceptible.

Rounded, convoluted galls, ranging in size up to several inches, usually are formed at the soil line on most susceptible plants (Fig. 35 and 36). On euonymus, however, galls may be formed anywhere along the stem. Low growing types, such as *E. acuta*, are frequently infected.

Figure 35. Crown gall on variegated euonymus (R. C. Lambe, VPI & SU).

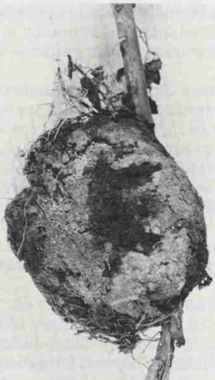

Figure 36. Close-up of crown gall on euonymus (R. C. Lambe, VPI & SU).

Bacteria enter the plants through wounds that are often the result of pruning and propagating procedures. Diseased plants may grow satisfactorily for some time but eventually become stunted and sections of the plant may die above a gall. The bacteria can survive in the soil for a period of 2 or more years.

The most effective control is the exclusion of the bacterium from the nursery. This can be achieved by using only healthy stock for propagation and sterilizing propagating tools frequently to remove bacteria. Plants should be grown in soilless media free of the bacteria or in fields with no history of crown gall infection.

Powdery Mildew

Powdery mildew, caused by the fungi *Microsphaeria penicillata* and *Oidium euonymi-japonici*, is prevalent on euonymus throughout the South and the Pacific Coast. The green euonymus 'Patens' is the most susceptible cultivar. Powdery mildew is rarely observed on variegated cultivars of *E. japonica*.

The fungus mycelium forms white powdery growth on upper and lower leaf surfaces, often when plants are crowded with insufficient air circulation. Unlike most fungi, mildew spores do not require a layer of free water on the leaf surface to germinate; high humidity is sufficient.

Provide for good air circulation around plants by proper spacing of plants. Use fungicides effective against powdery mildew if disease becomes severe.

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Holly Diseases

R. C. Lambe and J. J. McRitchie

Holly represents one of the most important groups of woody ornamentals grown in ornamental nurseries. There are numerous cultivars and hybrids with many different growth forms offered for sale. Hollies have originated in geographical regions with diverse climates and are frequently grown under conditions in the nursery that predispose them to disease, and therefore, it is not unusual for disease epiphytotics to occur. Some of the diseases that have been reported are restricted to single species of holly. In contrast, other diseases occur on several different species. In addition, certain cultivars of a species have been reported to be more susceptible than others. In general, Japanese holly is more susceptible to diseases than other hollies. Fungi have been reported as pathogens of holly more frequently than any other group of organisms attacking the leaves, stems and roots. Bacteria cause disease in landscape holly but they have not been particularly damaging under nursery culture. Nematode damage occurs more frequently in field production and landscapes than in soilless media in containers.

Diseases During Propagation

Foliar and soil-borne root rot pathogens can enter the nursery production cycle during propagation. High humidity and temperatures used during propagation are conducive to infection and disease. Some of the infected plants will be eliminated through death during propagation, but others will survive and serve as a source of the pathogen at later stages in the production cycle. Pathogens may be introduced into the propagation area through: 1) residue on the leaves and stems of cuttings; 2) contaminated rooting medium or propagation containers; 3) splashing or blowing up from the soil below or around propagation area; 4) water used for misting during propagation; or 5) unsanitary practices employed by personnel during propagation. Disease prevention starts with propagation. It is difficult to impossible to produce high quality saleable plants from low quality diseased liners. Propagation should be the most intensively managed practice in the nursery. Excellent sanitation is imperative.

Cuttings of cultivars of Japanese holly, American holly, and Yaupon holly are susceptible to *Rhizoctonia solani* during propagation. Defoliation begins 2 to 3 weeks after sticking. The stems are killed and a zonate leaf spot is a characteristic of the disease. Excessive water application by too frequent or too lengthy mist periods, lack of air circulation in the propagation area, excessive shade, leaving plants under mist too long, cuttings that are too large and cuttings stuck too close together will increase the frequency and severity of Rhizoctonia foliar blight. The disease occurs more frequently during the warm summer months. This same fungus causes web blight on larger plants in containers. Black root rot caused by *Thielaviopsis basicola* may occur during propagation. Black root rot will be discussed more completely under root diseases.

Container Culture

Under the intensive cultural practices, including high fertility levels, frequent irrigation and high plant density currently employed in the container nurseries in the South and Southeast, holly is frequently predisposed to disease. During a particular year it is not unusual for nursery plants to experience extreme cold, extreme heat and heavy rainfall of several days duration with resulting severe outbreaks of disease if the pathogen is available.

Containers of Japanese holly (*I. crenata*) grown under tightly crowded conditions of high humidity and high temperature are susceptible to Rhizoctonia web or thread blight. Dead leaves may be matted together or held suspended from the twigs by fungus hyphae, denser mats of which usually appear at the point of contact of diseased and healthy leaf blades binding them together (Color Plate VI, 5). Leaves have necrotic spots which may involve the entire blade. At maturity, the tan necrotic centers are surrounded by purplish-brown margins and the affected areas are brittle in texture, cracking and falling away under slight pressure. Diseased plants may be severely defoliated, but generally not killed. The lowest and inner leaves and twigs are the most susceptible.

Cuttings taken from such plants will introduce the disease into the propagation area. For more information see the section on web blight. During warm, humid weather, *Rhizoctonia ramicola* (possibly *R. solani*) attacks the leaves and twigs of holly in the landscape in Florida.

Several leaf spotting fungi have been reported on holly; most of them are of minor importance. *Cylindrocladium avesiculatum*, however, can cause a severe leaf spotting, defoliation and twig dieback of *Ilex cornuta*, *I. crenata*, *I. opaca* and *I. vomitoria* (Color Plate IV, 5). The disease first appears as tiny chlorotic spots which become purplish-black with a light green border. Defoliation is common and leaves may abscise with only a single spot. Twig dieback may follow heavy infection and defoliation.

In Florida, the fungus *Sphaeropsis tumefaciens* can cause witches' broom symptoms, galling and dieback on a number of ornamental hosts in the genera *Callistemon*, *Citrus*, *Nerium* and *Ilex*. *Ilex opaca* and *I. opaca* X *cassini* appear to be the most commonly affected hollies. Galls, ranging in size from swollen young twigs to large swelling on mature branches are characteristics of the disease. Several branches frequently arise from a single node and form a witches' broom. The fungus is easily spread by pruning tools and on cuttings. Stock plants should be inspected carefully before cuttings are taken, and any infected branches should be pruned well below any galls with pruning implements that have been dipped between cuts in a disinfestant.

When rooting or potting media are kept saturated with water for extended periods, *Pythium irregulare* and *P. vexans* can cause root damage in Japanese holly. They can be particularly damaging during vegetative propagation and liner stages. High moisture in the propagation medium must be avoided.

Japanese holly is susceptible to infection by the fungus, *Thielaviopsis basicola* causing a disease called black root rot. Once this fungus becomes established in the production area, it will be very difficult to eradicate. This disease has now been found in nurseries in several southern states, and may be more widespread in nurseries than is now recognized. Symptoms of the disease are stunting of foliage and root system and black lesions on root tips or elsewhere on roots (Color Plate IV, 1, 2, 3). Black lesions can best be seen by carefully separating the roots from the growing media and washing in water (Fig. 37). The foliage of infected Japanese holly exhibits marginal and interveinal chlorosis. In advanced stages of the disease, leaves are reduced in size, stem dieback occurs and the entire root system may be black and dead. The fungus produces conidia and chlamydospores on the surface and within infected root tissue. These structures aid in the spread of the fungus to nearby healthy plants and also serve as survival structures. Six cultivars of *I. crenata*, 'Helleri', 'Hoogendorn', 'Nigra', 'Green Cushion', 'Mobjack Supreme' and 'Hetzi' were moderately to highly susceptible. *I. vomitoria* and *I. opaca* are moderately resistant. English holly (*I. aquifolium*) and *I. cornuta* are highly resistant (Fig. 38).

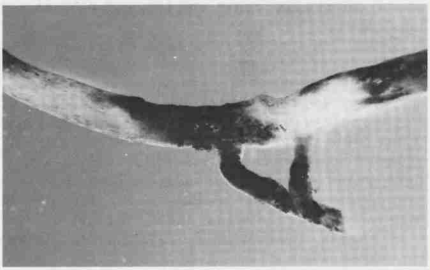

Figure 37. Microscopic view of Japanese holly feeder black root rot lesions (R. C. Lambe, VPI & SU).

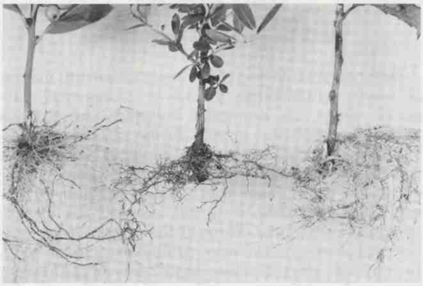

Figure 38. Black root rot on Chinese (left), Japanese (center) and English (right) holly (R. C. Lambe, VPI & SU).

Field Culture

During culture in the field, holly is susceptible to root rots and parasitic nematode attack. Root rot organisms may be present in the roots of susceptible plants in low populations with no apparent effect on plant growth. However, if the fields are not drained well or have impermeable subsoils, excessively heavy rainfall may result in prolonged saturated soil conditions and favor rapid multiplication of water mold fungi like *Phytophthora* spp.

On Chinese holly (*I. cornuta*), spot anthracnose develops on the upper surface of leaves, producing lesions on the shoots and scabby lesions on the berries. Leaf spotting and defoliation of *I. cornuta* 'Burfordi', *I. crenata*, *I. opaca* and *I. vomitoria* may also be caused by *Cylindrocladium avesiculatum*. Small chlorotic spots appear on the leaves turning purplish-black and enlarging to form circular lesions 10 to 15 mm in diameter. Mature lesions are circular, frequently zonate, 10 to 15 mm on *I. crenata* 'Helleri', *I. opaca* 'Savannah' and *I. vomitoria* 'Nana'. Leaves of English holly plantings in the northwest are susceptible to infection by *Phytophthora ilicis*. Rust of American holly caused by *Chrysomyxa ilicina* has been reported but is not ordinarily a serious problem.

Tobacco ring spot virus was reported in nursery grown *I. crenata* 'Rotundifolia'. Leaves on infected holly plants were permanently distorted although no observable reduction in plant growth occurred. Symptoms on older leaves consisted mainly of irregular leaf margins. This is probably of minor importance.

Canker and dieback, caused by the fungus *Gloeosporium* sp., occur on *I. cornuta* 'Burfordi'. Stem discoloration and defoliation occurred on terminal twigs. Sunken necrotic lesions were present in the cortical tissues of the twigs. A dieback has been observed on *I. crenata* especially where heavy pruning for shaping has been practiced. *Theilaviopsis basicola* black root rot, nematodes and other stresses have been observed in association with dieback.

Phytophthora cinnamomi is pathogenic on Japanese holly and causes dark streaks extending up the crown and lower stem. Similar symptoms have been reported on other woody host plants. This disease only occurs on holly growing under very poor soil drainage conditions. English holly growing in Virginia in the field under conditions of poor drainage is susceptible to infection by *P. cinnamomi.*

Nematode Diseases

Nematode diseases are probably the most important problem in field production of Chinese and Japanese holly. Symptoms caused by nematodes include yellowing, stunting, twig dieback and/or death of the foliage. Root systems may be stunted, with necrotic lesions and/or galled (Color Plate IV, 4). Hollies are attacked by four different nematodes that include *Criconemella xenoplax* (ring), *Meloidogyne* spp. (root-knot), *Pratylenchus vulnus* (lesion) and *Tylenchorhynchus claytoni* (stunt). These nematodes are found throughout the Coastal Plain and Piedmont sections of the Southeast. Ring and stunt nematodes are the most commonly occurring. The lesion nematode is found more in Piedmont soils and the root-knot nematode is usually found in areas that were once agricultural fields. The Chinese holly 'Burfordi' was the most resistant holly of several cultivars of Japanese and Chinese hollies tested to ring, root-knot and stunt nematodes.

Japanese holly 'Convexa' and 'Helleri' were damaged more severely than 'Rotundifolia' by root knot nematodes probably because of their more compact growth habit. As the number of root knot nematodes built-up in the soil, plant damage increased. Yaupon holly was tolerant to ring, root knot and stunt nematodes in field plot studies. In greenhouse studies, high numbers of ring nematodes damaged 'Helleri', 'Convexa' and 'Rotundifolia'. The plant response for holly cultivars and the four different nematodes are given in Table 14.

Table 14. Response of Cultivars of Chinese, Japanese and Yaupon Holly to Root Knot, Stunt, Lesion and Ring Nematodes in Field Plot Studies.

	Plant Response[1]			
Plant	Root Knot	Stunt	Lesion	Ring
Chinese Holly				
'Burfordi'	T	T	O	T
'Rotunda'	S	S	O	S
Japanese Holly				
'Compacta'	HS	T	T	S
'Convexa'	HS	T	O	S
'Helleri'	HS	S	O	S
'Rotundifolia'	HS	S	O	S
Yaupon Holly				
'Nana'	T	T	O	T

[1] Plant response rated as HS = highly susceptible with severe stunting, twig dieback and death, S = susceptible with some stunting but usually acceptable growth, T = tolerant with no apparent plant damage and O = combinations not yet tested.

Disease Prevention

Holly should be propagated from healthy stock plants and not from plants in the growing area. The cuttings should be rooted in pathogen free media, preferably in raised benches. Some fungicide application over the cuttings may be necessary to prevent root, stem and foliar diseases caused by soil-borne fungi in the genera *Rhizoctonia, Pythium* and *Phytophthora.* It may be necessary to treat the propagation and container media with fumigants or heat to eradicate pathogens before sticking cuttings. Consideration should be given to chlorination of irrigation water for propagation if the only available water source is pond water that must be recycled. Well water should not need such treatment. The containers used to propagate or grow in should be new or if reused, thoroughly disinfected before using. For more information see the section on Sanitation.

For field culture, poorly drained areas are likely to result in root rots caused by *Pythium* or *Phytophthora* spp. Nematodes are capable of causing severe damage to holly. Therefore, careful selection of pathogen-free fields or preplant fumigation are necessary to prevent root rots and nematode damage.

Containers in the field should be placed on crowned or drained beds. Water used to irrigate these containers should be free of plant pathogens. Close spacing or crowding may create the proper conditions for Cylindrocladium leaf spot or Rhizoctonia web blight. Providing good air movement between the plants will usually alleviate these problems. Foliar fungicides are suggested in some regions.

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Juniper Diseases

John Hartman

Twig Blight—*Phomopsis juniperovora*

Juniper twig blight disease (Phomopsis twig blight) occurs in most areas of the eastern United States where red cedar, common juniper and their horticultural varieties are grown. Although it is primarily a disease of seedlings and nursery stock, twig blight may appear on larger trees in ornamental plantings and on native cedars. This fungus disease normally becomes progressively less severe as trees become older. The twig blight fungus attacks arborvitae, cypress, Douglas fir, fir, hemlock, larch, redwood, white cedar and yew. Junipers reported to be resistant to twig blight disease under field conditions are listed in Table 15.

The tips of small twigs and branches affected by this disease first turn light green and then brown. In summer, the dead foliage turns ash-gray and small black spots (fungal pycnidia) appear on gray tissue. Cankers may be observed at the junction between dead and live tissue in the twig. Eventually, twig blight may be followed by a progressive dying back until an entire branch may be killed, decreasing the value of the tree or shrub. Survival of outplanted red cedar seedlings may be decreased if they have twig blight disease.

Phomopsis juniperovora overwinters in infected tissues as fruiting bodies and vegetative mycelium. During wet weather in the spring and in late summer or early fall, spores blown or splashed to healthy foliage germinate and penetrate the leaves. Infection only occurs when leaves are wet. The pathogen grows into twigs and causes cankers that eventually girdle the stem. The end of the girdled stem dies and the pathogen grows extensively in the dead tissue. Fruiting bodies can form on the dead tissue and produce spores as a source of inoculum for at least 2 years. Symptoms may develop within a few weeks after infection.

Juniper twig blight can be reduced in the nursery by observing the following suggestions: 1) provide adequate spacing between plants and rows to promote good air movement and rapid drying after rains; 2) avoid planting in heavy shade; 3) avoid handling plants when they are wet; 4) prune out infected branches during dry weather; 5) apply appropriate fungicide sprays to plants beginning with the new shoot development and again in the fall during periods of wet weather; and 6) use resistant varieties whenever possible.

Table 15. Junipers Reported to be Resistant to Twig Blight Disease Under Field Conditions.

Juniperus chinensis (Chinese juniper) femina, Iowa, keteleeri, pfitzeriana aurea, robusta, sargentii, sargentii glauca, shoesmith	***J. sabina*** (savin juniper) broadmoor, knap hill, skandia
	J. scopulorum (Western red cedar) silver king
J. communis (common juniper) aureo-spica, depressa, hispanica, hulkjaerhus, prostrata aurea, repanda, saxatilis, suecica	***J. squamata*** (Western red cedar) campbellii, fargesii, prostrata, pumila
	J. virginiana (Red cedar) tripartita
J. horizontalis (Creeping juniper) plumosa, plumosa aurea, procumbens	

Cedar-Apple Rust—*Gymnosporangium* spp.

Rust diseases occur whenever susceptible juniper and apple and its relatives are grown in close proximity to each other. In North America, cedar-apple rust (*Gymnosporangium juniperi-virginianae*) occurs on junipers throughout the eastern part of the United States and Canada. Several junipers and cedars in the genus *Juniperus,* including eastern and western red cedars and horizontal and savin junipers, are susceptible. Cedar-Hawthorn rust (*Gymnosporangium globosum*) occurs on eastern red cedar in the East and cedar-quince rust (*Gymnosporangium clavipes*) occurs on common juniper and eastern red cedar in the South. The existence of these juniper rusts requires infections of two types of host plants.

When heavy infections of cedar-apple rust occur, the juniper twigs and branches may be covered with hundreds of globose, deep brick-red to chocolate-brown galls ranging in size from one-fourth inch to 2 inches in diameter. The mature galls dry out, turn black and may remain on the tree for several years. Rust damage to junipers is not usually serious, unless several hundred galls are present on a single plant. The ends of the branches that bear the galls may die. Cedar-quince rust causes long fusiform branch swellings which bear the orange jelly-like tendrils of the fungus. The cankers resulting from the branch swelling can injure the infected juniper.

These *Gymnosporangium* species are rust fungi with a complex life cycle that involves production of several kinds of spores and involves infection of at least two host plants. See cedar-apple rust under crab apple for more details.

Infection of junipers in the nursery depends on: 1) the amount of rust development on nearby apple or related trees; 2) weather conditions in late summer when junipers may become infected, (dry late summer and fall weather frequently results in fewer galls being formed during the next 20 months); 3) locations of the apples with relation to the junipers, (the greater the distance depending on terrain and wind direction, the less infection). Rust diseases of junipers are primarily a problem in field production of large specimen plants and in the landscape.

The disease can be prevented by removing apples and related trees from the vicinity of the nursery. Because the disease usually does not cause heavy damage to junipers, this procedure is not normally suggested. Fungicide applications made at 2-week intervals during July and August will protect junipers from cedar-apple rust infection.

Table 16. Junipers Reported to be Resistant to Cedar-apple and Cedar-hawthorn Rusts.

Juniperus chinensis (Chinese juniper) femina, keteleeri, sargentii	***J. sabina*** (savin juniper) broadmoor, knap hill, skandia
J. communis (Common juniper) Aureo-spica, depressa, saxatilis, suecica	***J. squamata*** (Western red cedar) fargesii
	J. virginiana (Red cedar) tripartita

Root and Stem Rot Diseases

Some juniper cultivars and species are more susceptible to root rot than others. In general, root rot is more damaging on junipers produced in containers than those grown in the field. Western red cedar has been reported as a host of damping-off caused by the fungus *Rhizoctonia solani.* 'Andorra', 'Bar Harbor', *J. procumbens* 'Nana', 'Parsoni', 'Sargents' and 'Shore' junipers are susceptible to root rot caused by *Phytophthora cinnamomi.* Heavy losses of 'Shore' junipers in containers have been observed. Symptoms of juniper root rot resemble symptoms of general decline. The new growth may be light green and stunted. It becomes sparse and eventually the plant dies (Color Plate III, 6). Initially, small roots are killed and eventually lesions appear on larger roots (Color Plate III, 5). Identification of the particular causal agent requires laboratory analysis. Damping-off may result in failure of cuttings to root or in poor survival of rooted cuttings, following transplanting (Color Plate III, 4). Root rot must be avoided by cultural and sanitation practices. Fungicide drenches may also be useful.

Juniper Nematodes

Several genera of plant parasitic nematodes have been associated with common juniper and red cedar in the Southeast. These include: ring nematode (*Criconemoides*), spiral nematode (*Helicotylenchus*), sheath nematode (*Hemicycliophora*), lesion nematode (*Pratylenchus*) and dagger nematode (*Xiphinema*). Of these, only the root lesion nematode, *Pratylenchus penetrans*, has been reported to cause serious damage to juniper. In this case, red cedars were injured in a Great Plains nursery. 'Blue Rug' and 'Spiny Greek' juniper are severely stunted by lesion nematode. There are many juniper cultivars and only a very few have been evaluated for susceptibility to nematodes. Shore juniper appears to be tolerant to most nematodes.

The above ground symptoms of root lesion nematode infection resemble a nutrition deficiency or mild drought symptoms, whereby the plant is stunted and grows poorly. The root system is poorly developed and may lack feeder roots. Dead areas on many of the small roots are evident when roots are closely examined. Determination of the specific cause is difficult without culturing on selective media and nematode assay because root decay symptoms may be similar.

Nematodes should be avoided on junipers by taking cuttings only from plants free of nematodes. This is particularly important on prostrate growing types. Cuttings should be rooted in nematode-free propagation areas. Rooted cuttings should be transplanted to nematode-free potting media or fields known to be free of nematodes or the field should be fumigated prior to planting. If plants become damaged by nematodes, several systemic nematicides are available.

Leucothoe Diseases

J. T. Walker and R. K. Jones

Although there may be 50 or more species of these evergreen or deciduous shrubs, there are perhaps four which are native to North America and the Southeast: *Leucothoe fontanesiana*, *L. axillaris*, *L. racemose* and *L. populifolia*.

Drooping leucothoe, *L. fontanesiana*, formerly *L. Catesbaei*, is found from Virginia to Georgia and Tennessee. Another species, *L. axillaris*, the coast leucothoe, is very similar to *L fontanesiana* and found from Virginia to Florida and Alabama. A native plant called sweetbell (*L racemosa*) is a deciduous shrub found from Massachusetts to Florida and Louisiana. Its chief value as a landscape plant is in natural settings with shade and fairly dry soil. Recently several new cultivars of leucothoe have become popular nursery plants with a high level of disease susceptibility.

Powdery Mildew

Powdery mildew, caused by *Microsphaera penicillata*, was first reported on *L. axillaris* in south Georgia nurseries and may be economically important in landscape plantings where fungicides are not applied.

The first symptoms are yellow green spots with indefinite borders on the upper leaf surface (Color Plate VI, 2). The spots enlarge to 1 cm in diameter, become reddish purple and then are visible on the lower leaf surface (Color Plate VI, 3). Disease severity is intensified on plants held in overwintering greenhouses too long in the spring. Heavy leaf infection causes defoliation.

The disease can be controlled with regular fungicide applications. Because the disease is favored by high humidity, provide air circulation to decrease relative humidity around containerized plants.

Leaf Spots

Cylindrocladium leaf spot (*C. avesiculatum*) found on *L. axillaris* appears as brown spots with alternating light and dark rings which enlarge under high humidity to become a blotch. Leaf drop can become extensive and the disease can cause brown spots on twigs. Twig dieback then occurs as the twig lesions girdle the stem. Leaf spots can appear 48 hours after infection under warm humid conditions. No controls have been worked out but fungicides effective against *Cylindrocladium* disease should provide protection. Cylindrocladium is the cause of disease on numerous other plants, including azaleas and yaupon holly.

Another Cylindrocladium species, *C. scoparium*, was isolated from Rainbow leucothoe (*L. fontanesiana*) in North Carolina. Subsequent inoculations with this isolate caused leaf spots, wilting of foliage and by 20 days the fungus had invaded the stem. This fungus also causes a leaf spot, leaf drop and basal rot of cuttings on azaleas and in the past has caused extensive damage to azaleas in southern nurseries (Fig. 39). Allowing this disease to build up on leucothoe may provide inoculum for disease development on azaleas.

Figure 39. Cylindrocladium dieback on Rainbow Leucothoe (R. C. Lambe, VPI & SU).

Guignardia leaf spot, caused by the fungus *Guignardia leucothoes*, occurs on *L. axillaris* and *L. fontanesiana*. The 0.5 to 1.0 cm necrotic spots are surrounded by a margin of reddish-black and the lighter centers contain black specks (pycnidia) characteristics of the Guignardia blotch and leaf spot disease which occur on grape, horse chestnut, Virginia creeper and Boston ivy. Spores are spread from pycnidia via splashing rain or irrigation water. In the case of the disease on grape, spores germinate in 10 to 12 hours at 77° to 95°F (25° to 35°C) on wet leaves. Following penetration it may take 8 to 25 days before lesions appear on the leaves, first as small specks then as larger spots. Nothing is known about disease resistant cultivars of leucothoe.

Cercospora, *Phyllostica* and *Pseudomassaria* occur occasionally but are relatively non-destructive except under very moist and humid weather conditions.

Black Mildew

Black mildew caused by *Asterina diplodioides* occurs mostly in the humid climates of the South. This disease generally is referred to as black spot. The fungus is related to the powdery mildews but is black instead of white. It has been found on species other than the drooping leucothoe. Control measures are seldom undertaken on established plants. Fungicides may be appropriate in order to have saleable plants.

Leaf Gall

Leaf gall caused by the fungus *Exobasidium vaccinii* has been occasionally observed on *L. axillaris* in North Carolina. The disease is common on azaleas, andromeda, blueberry, huckleberry, rhododendron and other ornamentals. The bladder-like swelling of leaf and flower tissue in early wet spring is caused by a fungus but usually it is more unsightly than detrimental. Hand-picking the galls will reduce the amount of inoculum, but if many plants are infected, spraying with fungicides before buds open and after flowering sometimes is warranted.

Leucothoe is susceptible to root rot caused by *Phytophthora cinnamomi* but the disease is seldom a serious problem on this host.

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Ligustrum Diseases

Gary W. Simone

Ligustrum with its many species and varieties is an extremely familiar ornamental in the Southeast and may well be the most well-known shrub in the Florida landscape. There are few serious diseases that plague this mainstay of the landscaping industry. Root rots caused by *Phytophthora* and *Pythium* spp. are sporadic but can be serious in the nursery production area—especially during hot, moist weather or during excessively wet seasons. Details concerning Phytophthora root rots on ornamentals as a whole are available under the general discussion of this disease. (See general section on Phytophthora.) The two diseases consistently prevalent in nursery production are both leaf diseases—one caused by *Cercospora* spp. and the second caused by *Corynespora cassiicola.*

Three species of *Cercospora* cause a group of similar leaf diseases primarily (but not exclusively) on *Ligustrum japonicum,* the wax leaf or Japanese privet and *L. lucidum* the glossy leaf privet. New growth flushes are most susceptible. These fungi are active from spring through fall when adequate leaf moisture is available for infection. Distribution and severity of these specific diseases varies with locality within the Southeast.

Cercospora ligustri represents possibly the most aggressive of the three leaf-spotting fungi. First reported to occur in France, this fungus is now known to be common along the Gulf Coast, occurring on *Ligustrum lodense, L. amurense, L. ovalifolium* and *L. vulgare* as well as the Japanese and glossy leaf privets. Disease symptoms vary with the host affected. On the Japanese and glossy leaf privets, leaf lesions are circular in shape and range from 5 to 15 mm in diameter. Spots are faintly zonate with tan to brown centers and a wide reddish-purple margin. When disease is severe, leaf spots may coalesce killing extensive areas of leaf tissue. A slight, diffuse yellow halo may exist around these areas. The other *Ligustrum* spp. infected by *C. ligustri* develop smaller leaf lesions (2 to 5 mm in diameter) with typically tan-gray centers. In general, leaf spot development requires 10 to 12 days from infection with fungal reproduction on the upper leaf surface starting 14 to 21 days after infection.

Cercospora lilacis, although more common in Florida and other Gulf coastal areas than *C. ligustri,* is less damaging in the nursery. *Ligustrum japonicum* and *L. lucidum* are most commonly affected. Leaf spots are irregular to circular in shape with a range in size of 5 to 30 mm in diameter. Young lesions first appear as small yellow areas that are slow to enlarge. Mature leaf spots have dark brown centers with reddish to purple margins similar to those caused by *C. ligustri.* Lesions frequently coalesce and cause severe defoliation in the nursery. Symptom development and reproduction of the causal fungus is similar in time to that of *C. ligustri* except that *C. lilacis* reproduces from both leaf surfaces.

The third leaf spot disease, caused by *C. adusta,* is the least serious of the three *Cercospora* spp. This disease was first observed from Texas on *L. ovalifolium* (California privet) and is now known to be distributed eastward to Florida. Susceptible *Ligustrum* spp. are known to include *L. amurense, L. japonicum, L. lucidum, L. ovalifolium* and *L. vulgare* as well as other species. Leaf disease caused by *C. adusta* differs significantly from the other two *Cercospora* incited problems. Leaf lesions develop primarily on leaf tips or margins and occur in interior shrub portions or in well-shaded areas. Although several leaf spots may develop on a leaf, overall plant infection may be limited to 4 to 6 leaves throughout a plant causing minimal damage in the nursery.

Leaf lesions are circular to irregular in shape and range in size from 5 to 30 mm in diameter. Lesions have a depressed, brown center with wide reddish-purple to reddish-brown margins and are surrounded by a diffuse chlorotic band. Insects are often associated with the leaf spots and *C. adusta* may infect only after insect feeding wounds have been made in the foliage. The reproduction of *C. adusta,* as with *C. lilacis,* is from both leaf surfaces making control efforts difficult.

In the case of Cercospora-incited diseases of ligustrum, the best disease control option is prevention. The use of disease-free cuttings is extremely important due to the ease of disease spread under the mist propagation cycle. When production stock does develop one of the Cercospora leaf spot diseases, careful fungicide applications are required. Spray equipment should be calibrated for thorough penetration of blocks of infected plants. Sufficient pressure must be used to deposit fungicide on both leaf surfaces since both *C. ligustri* and *C. adusta* will reproduce from both leaf surfaces.

Corynespora leaf spot can cause heavy defoliation of the small leaf privet, *Ligustrum sinense.* This foliar disease is caused by the fungus, *Corynespora cassiicola* and is most severe during warm, moist periods of the production period. On variegated forms of *L. sinense,* leaf spots first appear as tiny, circular reddish spots that enlarge to a light brown lesion with a definite purple margin. The standard or non-variegated species are also susceptible but develop a lesion with a light to dark brown margin with a prominent yellow halo. Leaf spots will often coalesce and cause leaf abscission and severe defoliation on all the cultivars of *L. sinense.*

The fungus causes a common leaf spot disease on hydrangea as well as on such other crops as tomato, cucumber, soybean and cowpea but has not been shown to cross infect between *Ligustrum* spp. and the other plant species. Difficulty in control of this leaf spot disease with fungicides may necessitate alteration of irrigation regimes. Severely affected blocks of plants should be placed on a day watering cycle to limit the period of wet leaf surface and thus the likelihood of disease increases.

Palm Diseases

Ray Atilano

Stem and Leaf Necrosis

Leaf and stem necrosis is a common disorder of Chamaedorea palms, especially when grown in high relative humidity, moderate temperatures and under overhead irrigation. Plants are sometimes killed by the disease, but losses are usually due to the lower plant quality resulting from the death and removal of lower leaves.

The disease is caused by the fungus *Gliocladium vermoeseni* (Biourge) Thom. All *Chamaedorea* palms tested are susceptible to the disease, and include *C. cataractarum, C. elegans, C. erumpens, C. seifrizii* and *C. tenella*. The Areca palm, *Chrysalidocarpus lutescens* is also susceptible but is not usually affected as severely as chamaedorea palms.

Spores of the fungus are produced on dead plant tissue and are spread to healthy tissue by splashing water and air currents. The sheaths (petiole bases) and stem become infected at or near the nodes. Infection sites first appear as water-soaked spots less than a mm in diameter. Each infection site eventually becomes dark brown to black and may enlarge up to approximately 1 cm in diameter. Oozing of amber colored to black gum from infected tissue is frequently associated with the disease. As the sheaths die, they are colonized by the fungus which produces masses of conidia that appear as powdery, pink cushions on the exposed surfaces of the sheaths (Color Pate VI, 8). The fungus continues to progress inward through succeeding sheaths and may enter the stem. Leaves and affected sheaths dry to a straw brown.

The fungus is most active at 75°F to 81°F (24°C to 27°C), but activity is greatly reduced at temperatures over 86°F (30°C). Wounds are not essential for infection to occur, but injured tissue on sheaths or stems provides avenues of entry for the fungus and increases the amount and severity of disease. Injuries may occur during removal of senescent but-still-green sheaths. Free water on the plant surface is essential for the spores to germinate and penetrate directly into healthy tissue.

The incidence and severity of the disease in Florida is reduced during high summer temperatures. Practices that help to minimize the duration of moisture on plant surfaces will help to reduce disease development. Nursery operations that cause wounds should be avoided or modified to minimize the amount of injury to palms. The removal of chlorotic and senescent leaves during periods when temperatures exceed 30°C should minimize the chance of infection. If leaf removal is performed during periods favorable to the fungus, a fungicide application immediately after leaf removal may be warranted. The fungus is sensitive to benzimidazole fungicides. Preventive fungicide application programs should not rely solely on benzimidazole fungicides for control because of the possible development of resistance.

Bud Rot

Bud rots in palms are particularly serious because of their solitary bud nature. The disease affects palms in both container and field nurseries. Young palms which have buds near the soil line are most seriously affected, but under high inoculum pressure, buds 1 m or more high can also be affected. The older leaves are usually not affected until long after the bud has been killed, therefore, the disease is often recognized only after the most serious damage has occurred.

Phytophthora palmivora was shown to be the cause of bud rot in a *Washingtonia* palm field nursery. Other *Phytophthora* spp. have been implicated in bud rots of *Chamaedorea* and *Kentia* palms. Palm species affected include *Chamaedorea erumpens, C. seifrizii, Chrysalidocarpus lutescens, Cocos nucifera, Howea forsteriana, Neodypsis decaryii* and *Washingtonia robusta*.

Prolonged wet periods at 81° to 86°F (27° to 30°C) are most favorable for the disease. The pathogen may infect the plant directly but wounds near or below the soil line, such as those made when removing older leaf bases, increase the rate of infection. The succulent bud tissue is killed before that of the older petioles and leaves and the first visible symptoms are withering and dying of the central spear leaf and young unexpanded leaves (Color Plate II, 7). In some palms, such as *Chamaedorea* spp., a darkening of the tissue in the lower portion of the spear leaf accompanies the withering symptom. Recovery seldom occurs but may occur in field nursery-grown species such as *Washingtonia robusta*.

Contaminated particles of soil or potting media can be splashed onto the spear leaf or newly expanded leaves and the fungus may then infect the foliar tissue and spread into the bud. Dark, irregular, spreading lesions are evident on the expanded leaves when this occurs.

The pathogen is commonly found in soil and the chapter on soil-borne diseases and Phytophthora diseases should be consulted for details of how this pathogen may be harbored and spread in a nursery. In container nurseries, the disease is more prevalent where the pots are placed directly on the ground or subject to frequent shallow surface flooding. Practices that help to reduce bud rots include the use of pathogen-free soil, isolation of pots from the ground and minimizing the wetness of the soil and environment. New systemic fungicides specific for *Phytophthora* spp. can be of great value in preventing and reducing the spread of the disease.

Leaf Spot

Damage due to leaf spots is generally more critical in palms grown for foliage than in palms grown for landscape use. The principal difference between the two types of palms is that palms produced for landscape use have a much higher tolerance for leaf spots, in that they may incur a greater number of spots without serious economic loss or damage to plant health than do palms grown for foliage.

Leaf spot pathogens of palms include many genera of fungi. Fungal genera that have been associated with leaf spots are classified under *Bipolaris, Cercospora, Cylindrocladium, Dreshlera, Exosporium, Graphiola, Helminthosporium* and *Phaeotrichonis*. Little has been published showing which of these fungi are most important in the Southeast. In southern Florida the genera most frequently encountered on foliage palms are *Bipolaris, Dreshlera, Helminthosporium* and *Phaeotrichonis*. Other areas may have these or other fungi as important leaf spot pathogens, depending on the climate and plant species.

Spores of leaf spotting fungi are commonly transported by air currents from dead plant material in and around the nursery to the palm leaves. The spores may remain dormant, but alive, for days to weeks on the leaf surfaces. Infection only occurs when moisture is present on the leaf surfaces for several hours during moderate to warm temperatures (72° to 85°F).

Little information is published on the role of other physical factors and cultural practices in the development of leaf spot diseases. However, some general comments may be useful. Leaf spot severity frequently increases during or following growing conditions that have been stressful for the plant. For example, stresses, such as low fertility or drought, seem to precondition palms to increased susceptibility to leaf spots. Suboptimum light levels may interact with fertility and soil moisture in increasing susceptibility of palms to leaf spots. The influence of growing conditions on the susceptibility of palms to leaf spots is clearly a subject that needs research.

Control of leaf spots in palm nurseries can be achieved by basic disease control methods. Diseased plants and plant debris should be removed from the nursery. Leaf wetting should be minimized, and the drying of leaf surfaces should be promoted by timing of irrigation for quick drying. Wide alleys must be established between plant blocks with wide plant spacing to encourage air movement. Provide fertility, water and light for good plant growth and the judicious use of fungicides will continue to be of value.

A leaf spot of the fish tail palm *Caryota mitis* is caused by the bacterium *Pseudomonas alboprecipitans*. The lesions in the leaves are usually linear (2 to 50 mm long), brown to black and sometimes surrounded by a yellow margin. The disease is most severe when the leaves are wet for several hours. Chemical control of bacterial diseases of foliage plants is only partially satisfactory. Cultural practices that are effective include pruning off diseased foliage and using methods that promote leaf drying.

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Photinia Diseases

Donald J. Blasingame

Photinia (*Photinia Fraseri*, *Photinia glabra* and *Photinia serrulata*) is a large plant with large, upright, stiff growth. The leaves of *P. Fraseri* and *P. glabra* are bright red when young and dark green when mature. In the nursery, photinia is grown both in containers and in the field.

Powdery Mildew

Powdery mildew, caused by *Sphaerotheca pannosa*, is a serious disease on *Photinia serrulata* and is a very common disease on a number of woody ornamentals. The disease weakens the plant by attacking the bud, young leaves and growing tips. The disease first appears on young leaves as slightly raised blister-like areas that soon become covered with a grayish-white powdery growth containing spores (Color Plate VII, 6). The young leaves do not expand normally, but become curled and distorted. On older leaves, large white patches of the fungus appear but cause very little distortion. Tiny black fruiting bodies of the fungus are often embedded in the white patches, especially late in the growing season.

The disease can spread rapidly in the nursery under conditions of high humidity (cool nights—warm days), crowded conditions and poor air circulation. Spores are easily blown about by the wind and scattered by overhead watering. Powdery mildew is reduced through wide spacing of containers to provide good air circulation. Fungicides should be applied to the new growth and repeated to protect the young leaves. *Photinia Fraseri* and *P. glabra* are resistant to powdery mildew.

Leaf Spot

Entomosporium leaf spot, caused by *Entomosporium maculatum*, is generally distributed on pear and quince, widespread on amelanchier, and is sometimes found on apple, Japanese quince, medlar, mountain-ash, Siberian crab, cotoneaster, pyracantha, India hawthorne, loquat and photinia. Very small purple spots appear on leaves in the spring. The spots enlarge to a brownish circular lesion one-fourth inch or less in diameter, with a raised black fruiting body in the center of each spot (Color Plate II, 6). If spots are numerous on the leaves, extensive defoliation will result. Damage in the nursery renders the plants unsaleable.

Twig lesions appear on the current season's growth about midsummer; indefinite purple or black areas coalesce to form a canker. Primary spring infection comes from conidia produced in these twig lesions. High humidity, cool temperatures, crowded conditions and overhead watering provide ideal conditions for disease development. This disease has been more damaging in container grown plants than in field production or in landscape plantings.

Many nurserymen will attempt to prune defoliated plants for later sale. However, unless this procedure is coupled with a tight schedule of fungicide sprays, the disease will reappear. Coupled with a preventive disease program of good sanitation is wide spacing of plants. Photinia cutting blocks (field grown or landscape plants) should be free of leaf spot. In the Southeast, fungicide sprays should begin before new growth starts, since this is when the spores are first released. If Entomosporium leaf spot has been a problem on mature container grown plants, several additional fungicide applications should be made until new leaves have matured. Several applications in the fall may be helpful every 10 to 14 days. Fungicides should not be needed on field grown plants if the liners are disease-free and isolated from other sources of the pathogen. Once this disease has become established in a nursery, control can be difficult. Do not purchase liners to grow-on if they are diseased.

Other leaf spots include *Gloesporium* spp., *Cercospora* spp., *Phyllostica* spp. and *Pestalotia* spp. Although these fungi are widely distributed and occur every year, they cause only minor damage in the nursery. These fungi attack both old and new growth with leaf spots of various sizes and coloration on both the upper and lower surfaces of the leaf. Under severe conditions, some defoliation may occur. Conditions for the development and spread and control of leaf spots are similar to those for Entomosporium leaf spot.

Fire Blight

The fire blight bacterium, (*Erwinia amylovora*), is worldwide in distribution and has a host range including many rosaceous fruit and ornamental plants.

Fire blight normally occurs on the young succulent leaves and twigs of photinia during the spring months. Twig infections progress very rapidly under warm, moist conditions. The young growth appears to have been scorched by fire and may exhibit the typical "shepherd's crook." The bacteria enter the plant through stomata and hydathodes of the young leaves and progress inward by way of the major veins. Spread of the organism is primarily by wind and splashing water. For more information on fire blight, see crab apple.

In the nursery, conditions that produce excessive succulent spring growth should be avoided. Do not overcrowd plants, but if infection occurs, carefully prune out diseased parts and limit overhead watering. A preventive spray program should be considered only as a last resort.

Pittosporum Diseases

William H. Ridings and Calvin L. Schoulties

Angular leaf spot of *Pittosporum tobira* (variegated and green forms) caused by the fungus *Cercospora pittospori* is characterized by angular, vein delimited, chlorotic spots varying from 1 to 5 mm in size that may coalesce to cover much of the leaf. On the non-variegated form of *Pittosporum*, the leaf spots are yellow to dull brown on the upper leaf surface whereas they are gray to light-tan on the lower leaf surface. On the variegated form the leaf spots differ only in that they are tan to yellowish-tan on the upper leaf surface. Sporulation by the fungus occurs profusely on the lower leaf surface. High humidity and warm temperatures favor disease development. The disease is considered more unsightly than damaging to the plant; however, defoliation may occur under severe disease stress. Fungicidal protection has been effective to control this disease.

Alternaria leaf spot, caused by the fungus *Alternaria tenuissima*, varies from young chlorotic spots (0.5 mm to 3.0 mm in diameter), to older circular to subcircular spots (1 to 6 mm in diameter) with a diffuse yellow halo at the margin of each spot. Both the young and older spots have a necrotic area in the center. Leaf spotting occurs more frequently on the young leaves resulting in some leaf discoloration and an unsightly appearance of the leaves. Fungicidal protection has been the best control.

Silky thread blight caused by the fungus *Rhizoctonia ramicola* affects the leaves, twigs and petioles. Leaf infection is evidenced as small necrotic spots with indefinite margins. These spots enlarge slowly to tan lesions which may cover a large portion of the leaf blade; the lower surfaces of the infected leaves are covered with mycelium of the fungus which resembles a web. Infected leaves may become matted and hang from the twigs. High temperature 75° to 98°F (24° to 36°C), and humidity (75 percent) are favorable for disease development. The fungus may overwinter as long as 7 months in diseased tissue. It does not survive in the soil. Protective fungicides have been used for control.

Stem galling cankers are caused by the fungus *Nectriella pironii* (Imperfect state = *Kutilakesa pironii*). Galls are a subspherical to elongate, corky and roughened proliferation of callus tissues which develop to surround the stem. The fungus is frequently evident on and in the crevices of the galls as orange-colored sporodochia which can be viewed with a 10 X hand lens.

The fungus initiates infection primarily through pruning wounds. Pruning tools may transmit the disease organism. Control consists of roguing and discarding severely infected plants. Pruning judiciously lightly-infected plants and applying a fungicide-miticide combination have been suggested for control.

Crown gall, caused by the bacterium *Agrobacterium tumefaciens*, usually occurs at the basal stem area and on the roots, but may be found on aerial portions of the stem. Galls are spherical to subspherical, swollen, smooth proliferations which do not surround the stem. Infection is primarily through wounds contaminated by dirty pruning tools. The bacterium can survive in the soil. Diseased plants and soil should be rogued and removed from the nursery area.

Roughbark is a disease considered to be caused by a virus although it has not been proven conclusively. Symptoms consist of elongate (up to 10.0 cm), roughened and fissured stem swellings which may appear anywhere on the plant but are more often found on the older portions of the stems (lower branches and main stem). Plants are often dwarfed and unthrifty. Leaf symptoms indicative of a virus infection have not been observed in the southeastern United States.

The disease-causing organism is transmitted in cuttings from infected plants. Control consists of roguing infected plants and obtaining propagative stock from healthy plants.

Stem and root problems caused by the fungi *Rhizoctonia solani, Sclerotium rolfsii, Pythium* spp. and *Fusarium solani* have been observed. Infected roots show sloughing of the root cortex. Symptoms of infection for *R. solani* have been noted under *Podocarpus*. Infection by *S. rolfsii* is evidenced by white mycelium and mustard seed-like sclerotia at the base of the stem. Detection of *Pythium* sp. and *Fusarium* sp. is best distinguished by cultural characteristics in a plant disease diagnostic laboratory. Controls consist of sanitary procedures and protective fungicides.

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Podocarpus Diseases

William H. Ridings and Calvin L. Schoulties

Diseases of *Podocarpus* spp. are caused primarily by root rotting fungi such as *Rhizoctonia solani*, *Pythium splendens*, *Pythium* spp. and *Phytophthora* sp. which may attack plants at the seedling, rooted cutting and containerized plant growth stages. Young plants show poor vigor and when removed from the culture medium show sloughing of the root cortex. The fungus *Rhizoctonia solani* may develop mycelium partially up the stem. Culture-medium particles may become trapped in this mycelium which results in a dangling effect of the particles when an infected plant is removed and examined. Diseased specimens should be submitted to a plant disease diagnostic laboratory to confirm the presence of one or more root rotting fungi.

No important problems with foliar plant pathogens have been noted on *Podocarpus* spp. in nursery production.

The best control of the root rot diseases consists of strict sanitary procedures in the propagative bed (such as clean cuttings, seeds and culture media). Fungicide drenches have been used where root disease problems are threatening.

Rhododendron Diseases

D. M. Benson

Rhododendrons are becoming an increasingly important nursery crop in the Southeast. Diseases and their control can be a major limiting factor for many growers. Hybrid rhododendrons, heavily fertilized, with overhead irrigation in containers, generally have more disease problems than those grown in field soil in the mountains and native species grown as cutbacks.

Phytophthora Root Rot

Phytophthora cinnamomi as well as several other species of Phytophthora including *P. cactorum*, *P. citricola*, *P. cryptogea*, *P. gonapodyoides*, *P. lateralis* and *P. megasperma* are reported to attack rhododendron. The most important of these are *P. cinnamomi*, *P. citricola* and *P. cactorum*. Phytophthora root rot develops on both container and field-grown plants when environmental conditions are favorable and spores are present.

Rhododendrons are more susceptible to Phytophthora root rot than most other woody ornamentals. Plants may be killed during propagation phases as well as during the "growing-on" stage. Obviously, small plants with few roots will die more quickly than larger plants when infected by *P. cinnamomi*. In the landscape, large plants may "decline" over a several-year period before completely collapsing.

Infected cuttings in propagation typically exhibit a noticeable downward droop or wilting of the leaves, a failure to initiate new shoot growth as well as a dull green cast to the foliage. Examination of roots will disclose a reddish-brown color of roots rather than the white appearance of young, healthy roots.

Containerized rhododendrons that develop Phytophthora root rot will show several symptoms. New shoot growth may or may not develop. If new shoot growth does develop, leaves may permanently wilt. On plants that do not develop new shoot growth, leaves may droop and become dull green as in the propagation house (Color Plate I, 7). Roots will be reddish brown in color (Color Plate I, 8). By the time foliage symptoms develop, a reddish-brown stem discoloration may be evident at or just below the soil line. In more advanced stages, the stem discoloration may progress up the stem several inches above the soil line.

Control of Phytophthora root rot of rhododendron follows the general recommendations made in the "General Disease" Section. In addition to the cultural and chemical control measures, use of resistant varieties is important. Hoitink and Schmitthenner tested 336 hybrids and 198 species of rhododendron for resistance to root rot. The most resistant hybrids were 'Martha Isaacson', 'Caroline', 'Professor Hugo de Vries' and 'Red Head'. Fourteen other hybrids including 'English Roseum' were moderately resistant.

Twelve species proved resistant to *P. cinnamomi*. One of the most susceptible rhododendrons was 'Purple Splendour'.

Even on resistant cultivars, feeder roots may become infected. In well-drained media, new roots regenerate from the crown. Under poorly-drained soil conditions even the most resistant hybrids may succumb to root rot.

Phytophthora Dieback

Phytophthora dieback has been identified since the 1930s and was reported to be caused by *P. cactorum*. It has only been in the last few years of the 1970s that this disease became important in southern nurseries. Recently several species of Phytophthora have been reported to cause rhododendron dieback but the most important are *P. cactorum*, *P. citricola* and *P. parasitica*.

Symptoms of Phytophthora dieback first appear as brown irregular-shaped lesions on leaves or as a brown discoloration extending up and down the stem (Color Plate III, 1). Infection typically occurs at the margin of the lower leaf surface, progresses through the leaf to the midrib and then through the petiole into the stem (Color Plate III, 2). Eventually the pathogen may spread to other leaves via growth through the stem. Infection only occurs on succulent current season's stems and leaves, but slowly moves into older portions of the plant. On first year plants, the pathogen may move from a new leaf infection to the soil line in 7 days.

Initial infections develop when overwintering inoculum is splashed up to the new foliage in early summer. Symptoms will occur within 1 to 2 days after infection. Secondary spread of inoculum occurs when infected tissue remains wet overnight. Then spores develop and are splashed by subsequent overhead irrigation to adjoining plants. Optimum temperature for sporulation and infection is between 78° to 86°F (25° to 30°C). Overnight periods of wetness are necessary for spore formation. Splashing water in the form of either rain or irrigation is needed for spread (Fig. 40).

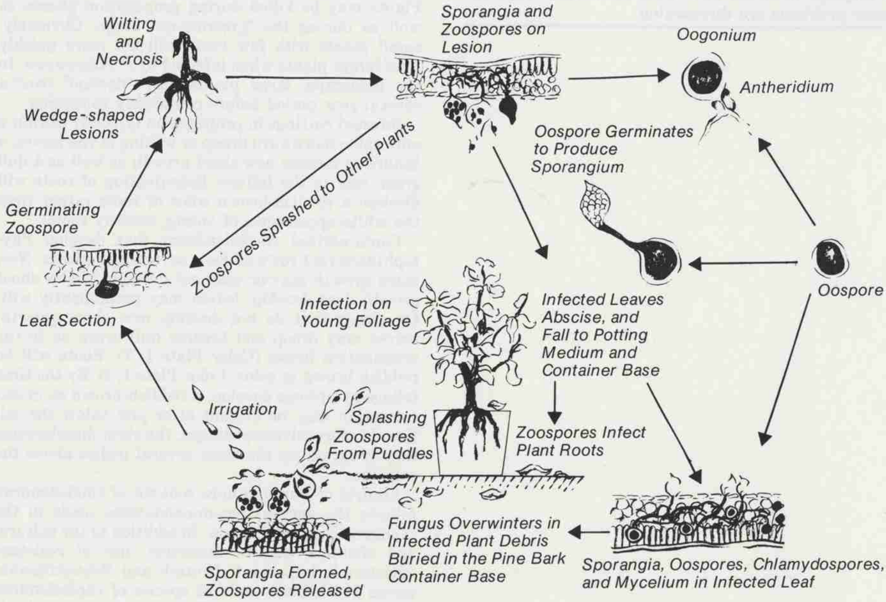

Figure 40. Illustration of disease cycle of dieback of Rhododendron caused by the fungus *Phytophthora parasitica* (Cheryl Kuske, NCSU).

Control of dieback depends on cultural practices to avoid or minimize the disease and fungicides to prevent infection. During hot summer months, growers should avoid late afternoon or early morning irrigations that would favor spore formation or spread newly-formed spores. Irrigation should be done at midday after the dew has dried off the foliage so any spores will dry out in the afternoon and die. If the foliage dries off before nightfall the production of spores will be greatly reduced. All infected shoots should be pruned out on a twice weekly basis. Excess fertilizer, particularly nitrogen will result in plants that are very succulent and hence highly susceptible to dieback. Excess shade also increases disease incidence.

Several fungicides have been tested and found effective for dieback control. These materials must be applied before infection occurs so a regular fungicide preventive program is necessary during summer months. Check with your state extension specialist for the fungicides available in your area.

Resistance of hybrid rhododendron cultivars to dieback has not been fully tested. However, under greenhouse conditions, 'Jean Marie de Montague' and 'Madam Mason' were not resistant. Under epidemic conditions in one nursery, 'Roseum Elegans' and 'Roseum Pink' were highly resistant while 'Scintillation' and 'Chinoides White' were highly susceptible.

Botryosphaeria Dieback

Dieback and canker of rhododendron caused by *Botryosphaeria ribis* (*B. dothidea*) can be a serious disease. Infection is generally limited to wounds on stems where subsequent cankers and dieback develop. Symptoms are reddish-brown to black sunken cankers that girdle stems. Cankers may develop from infection of natural wounds such as leaf scars or pruning wounds up to 2 months later. Stem discoloration is confined to the bark initially and later spreads to the wood and pith. Leaves on affected stems droop and roll inward along the midvein. Plants may be more susceptible during periods of drought stress. The disease is more likely to occur in field grown or landscape plants.

Botryosphaeria dieback may be confused with Phytophthora dieback of rhododendron. *Botryosphaeria* is generally slower to develop cankers, requiring 135 days to colonize 94 mm of stem and it is usually restricted to stems. On the other hand, *Phytophthora* spp. spread from stems into leaves producing a wedge-shaped zone of discoloration down the leaf.

Pruning tools were implicated in the spread of *Botryosphaeria* spores; hence disinfecting solutions should be used to treat tools. No other control measures, including resistant cultivars, cultural practices and fungicide sprays, are known for Botryosphaeria dieback of rhododendron.

Cylindrocladium

Under nursery conditions, rhododendron is occasionally attacked by *Cylindrocladium scoparium* and *C. theae*. These fungi cause dark brown to black lesions on leaves, stems and petioles. This disease is favored by warm temperatures and excessive water. The disease can be controlled by good water management, prompt pruning and fungicide sprays. For more information on this disease, see *Cylindrocladium* under General Diseases.

Nematodes

Information on rhododendron decline caused by plant-parasitic nematodes is incomplete. Ten different genera of nematodes were associated with rhododendron in one New England survey and six different genera were found on southern native rhododendron in a southeastern United States survey. The effect of individual nematode species on hybrid rhododendron is not documented, although azaleas are susceptible to the stunt nematode.

In general, production in geographical areas that are damp and cool during summer months would discourage nematode activity.

Leaf Gall

In the springtime, enlarging shoots of rhododendron may develop a thick swollen gall caused by the fungus *Exobasidium vaccinii*. Leaf gall only affects new growth and hence will not kill the plant, although general plant vigor may be reduced. Galls may become quite large and convoluted and eventually they develop a whitish cast. Spores are liberated by air currents from the whitish layer on the gall. These spores land on newly-formed buds for next year's growth where they infect or remain dormant over winter months. In the spring as the bud expands, the fungus grows at a rapid rate producing the gall. Local folks apply the term 'pinkster galls' to this growth on native rhododendron. Leaf gall is common in field grown plants, stock plants, native plants and landscape plants.

Control of leaf gall is routinely accomplished by picking off and burying the galled tissue. This is an annual job on highly susceptible cultivars and species. Since the disease causes little real damage, fungicide sprays generally are not necessary. If needed, fungicide sprays should be applied as the whitish layer begins to develop on galled tissue. On plants to be protected, sprays should continue at 10 to 14 day intervals until all galls have dried up and are no longer producing spores.

For more information on Petal blight, see the section on azaleas.

Rose Diseases

Norman L. McCoy and George Philley

There are basically two systems of rose production, field and container grown. The major disease problems common to both systems will be addressed in this section.

Roses are susceptible to a number of diseases, the control of which is not difficult if the nature of the diseases is understood and if production and preventive measures are applied systematically and continuously.

Another aspect of disease prevention is the service provided by nursery inspectors of regulatory agencies that help rose producers detect diseases by routine inspections. For example, if crown gall is detected in a rose-growing area, infected plants may be quarantined and properly disposed. This system works well to prevent the spread of this destructive disease.

Black Spot

Black spot, caused by the fungus *Diplocarpon rosae*, is probably the most serious rose disease. Spores of the black spot fungus are dispersed by splashing raindrops and not by wind. The presence of free moisture is necessary for black spot infection to occur. The primary symptoms are irregularly-shaped black spots with feathery edges often surrounded by a yellow halo on the foliage (Color Plate VII, 4). Infected leaves eventually drop off and this will be more severe under warm temperatures rather than cool temperatures. Heavy infection causes excessive and premature defoliation, reducing the carbohydrate content of canes and roots which in turn reduces the amount of subsequent foliage and flowering parts. In addition to leaf spots, infection also occurs on petioles, twigs and canes. The fungus overwinters on leaves and stem infections.

The fungus invades the bark tissue when it is tender and is often the overwintering place of the fungus. This is probably the most important aspect of perpetuation of the black spot fungus from one season to the next. The bark infections do not seem to hurt the canes but do serve as a source of spores to recreate the disease. With a good spray program, these infection sites appear to degenerate in a year or two. Pruning infected canes during the late winter, before new growth starts, will help destroy infected plant parts and aid in preventing the occurrence of the disease the next growing season.

One of the main things leading to good control of black spot is to start spraying early when foliage buds swell and continue until frost or freezing weather. Another factor in effective control is to apply enough spray to get good coverage of all the foliage. With some fungicides a wetting agent will improve coverage.

There are many fungicides cleared for black spot control. While most fungicides are very safe, plant damage may occur when some are used under certain conditions and on certain varieties. Red spotting on the foliage results from copper sprays and dusts when applications are made in cool, cloudy weather. Another phytotoxic symptom is marginal burning which can occur when dusting sulfur is applied during hot weather.

The fungicide or fungicide mixture that is selected should be applied to give complete coverage on the upper and lower leaf surfaces. Applications should be repeated at approximately one-week intervals.

Powdery Mildew

Powdery mildew is caused by the fungus *Sphaerotheca pannosa* and the infection results in a white powdery substance on the leaves, buds and twigs causing them to be distorted and dwarfed (Color Plate VI, 7). With serious infections, the tips of canes may be killed. Frequently, the unopened buds are white with mildew before the leaves are affected to any great extent. The disease is more likely to occur during cool, dry conditions and can spread rapidly since a complete life cycle can occur in 72 hours. Often, infected buds do not open. Petals, sepals and receptacles of the flower buds are also subject to attack.

Powdery mildew is usually not a season-long problem like black spot, but rather develops when weather conditions permit. The secret to control lies in beginning fungicide applications as soon as symptoms are noted. Frequent sprayings will keep the disease in check and protect new growth. Only a few of the black spot control fungicides will control powdery mildew. Benomyl has not given consistent control in field-grown roses. It appears tolerant strains of the fungus develop after repeated applications.

Canker

Roses weakened by black spot and powdery mildew are more susceptible to dieback and canker caused by the fungi *Crytosporella umbrina* and others. Organisms invade the stems and cause cankers (dead tissue surrounded by living tissue) to form. Canes die from tip downward, often starting in the flower stems. Diseased wood turns brown or black and is somewhat shriveled. Infected limbs and twigs should be pruned well below the diseased tissue.

The best approach in controlling dieback and canker is to improve the vigor of the plants and use fungicides for the control of other diseases.

Rust

Occasionally rust, caused by the fungus *Phragmidium* sp., becomes a problem on roses. Infection sites result in small, orange or yellow pustules on green portions of the plant (Color Plates IV, 7). In early spring, these masses may be so inconspicuous as to be unnoticed. In the late summer or early fall, the spots change and black pustules appear, frequently in the same affected areas. These pustules overwinter within the leaf and stem tissue after the leaves have fallen and later produce the spores that cause the spring infection. Young stems and green parts of the flower may also be infected. Severe spring pruning, the use of a dormant spray and weekly applications of an effective fungicide will help control rust.

Crown Gall

Woody-type galls, caused by the bacterium *Agrobacterium tumefaciens*, form on the lower stem and root tissue. If this tissue encircles the lower stem or main roots, the plant may be killed. As the galls enlarge, they become woody with a rough and irregular surface (Color Plate VIII, 5). Aerial galls can develop, but most are found at or just below the soil line. Galls range from pea-size to larger than one foot in diameter.

The crown gall bacterium enters plants through wounds, such as those arising from cultivation, transplanting, wind damage or insect injury. Wounds that have healed beyond a certain point are no longer susceptible to invasion. Pruning off galls is not effective since the gall tissue can reproduce itself. Chemical control with antibiotic drenches has shown some promise; however, they are not practical at this time.

The following production practices will assist nurserymen in keeping crown gall under control:

1. Inspect incoming roses for crown gall before replanting.
2. Remove and destroy all infected or weakened plants. Dig up as many roots as possible.
3. Avoid wounding roses while mowing or cultivating.
4. Keep roses in an active growing state with proper fertility and watering.
5. Control root-feeding insects.
6. See general section on crown gall for information on biological control.

Nematodes

Root knot nematodes are the most common on roses. They form knots or galls on the roots which limit root development and movement of water and nutrients in the vascular system. Dagger nematodes cause similar damage, but the swellings are more noticeable at the root tips. The result of heavy nematode damage is a less vigorous, possibly stunted plant.

If plants are to be grown in infested soil, treating the soil with a sterilant or fumigant chemical should precede planting. Where plants are established, treating with a systemic nematicide may be helpful.

Not all soil nematodes are harmful. Some live in the soil close to plant roots and cause no damage. A few are actually beneficial, feeding on such harmful pests as Japanese beetle grubs. Only an expert nematologist can determine species and decide which are responsible for a plant's ill health.

In determining the presence of nematodes and the extent to which they may be harmful, soil samples can be collected and submitted to your Experiment Station or Extension Service for diagnosis. Roots should be dug with some surrounding soil and placed immediately in a plastic bag to prevent drying out and mailed as soon as possible.

Viruses

Several different viruses infect roses. Symptoms include mosaic patterns, chlorotic veins and streaks. Some patterns may appear to be rather artistic. Most rose viruses are spread from plant to plant by graft transmission rather than by insects which is common for most viruses. Serious damage is not usually observed; however, stunting and poor growth can occur. Grafting or budding wood should be taken from known virus-free plants. There are no chemical controls for viral diseases.

Rose mosaic is the most common virus in the United States and other countries where roses are grown. This is an old disease which occurs in nurseries, rose gardens and commercial field plantings. Depending on the variety and on variations of the virus, rose mosaic symptoms range from a general chlorosis, to vein clearing or banding, distinct rings or intricate line and mosaic patterns. Plants infected with rose mosaic may be somewhat stunted and, in colder climates, may suffer greater winter injury than healthy plants.

Rose Streak—This is a suspected virus disease of roses occurring primarily in the eastern states. This disease is thought to be transmissible by grafting and seems to affect only roses.

Rose Rosette—This disease is reportedly transmitted by a mite and the viral etiology has not been proven.

Rose Leaf Curl (RLC)—This disease was reported in California in 1976. The symptoms resembled those of rose wilt or dieback, a disease not known to be present in the United States although it is found widely in Australia and New Zealand.

Potential Cell and Tissue Culture for Rose Virus Control. Virus-free plants of roses have been obtained by culturing apical meristems or shoot tips. If the growing tip is excised and grown in a sterile medium, a virus-free culture can be obtained. Once pathogen-free tissues are established, plantlets derived from such cultures are likewise pathogen-free. The full potential of tissue culture has yet to be exploited with roses, but it appears that rose growers and breeders will benefit in the future as these techniques are adapted for practical application.

Botrytis Blight

Botrytis blight and canker caused by the fungus *Botrytis cinerea* causes flower buds to droop and fail to open. Botrytis blight occurrence is directly related to wet weather and high humidity. As blossoms decay from infection, a light brown fungal growth appears. This fungal growth aids in identifying the disease. Just below the flower head, there is sometimes a smooth, slightly sunken, light tan to grayish black lesion extending down the stem (Color Plate VI, 6). This can be the most damaging phase of the disease.

All infected blossoms should be cut and destroyed as soon as they begin to droop or appear blighted. The black spot control chemicals will usually control botrytis blight.

Leaf Spot

Leaf spot caused by the fungus *Cercospora rosicola* results in small leaf spots with brown to black centers and a purplish border. It is particularly noticeable on multiflora understock which is usually not sprayed with fungicides. Defoliation is much less than that associatedwith black spot.

Many more diseases have been reported on roses. Some that may be observed are: Verticillium wilt (*Verticillium albo-atrum*), cotton root rot (*Phymatotrichum omnivorum*), southern blight (*Sclerotium rolfsii*), downy mildew (*Peronospora sparsa*), mushroom root rot (*Clitocybe tabescens*) and anthracnose (*sphaceloma rosarum*).

Taxus Diseases

John Hartman

Taxus is more widely grown in the upper southern states. It is grown mainly in the field and sold as balled and burlapped plants.

English yew (*Taxus baccata*) and its varieties, as well as Japanese yew (*Taxus cuspidata*) nursery stock and ornamental plantings, are highly susceptible to Phytophthora root rot. Heavy losses may occur in heavy and poorly drained soils. The foliage of infected plants often turns yellow at the growing tips, followed by general yellowing, wilting and death of the plants over a period of several months. The entire fibrous root mass becomes rotted and dark brown necrosis extends up into the stem a few inches beyond the root collar. For more information, see the general section on Phytophthora root rot.

When a yew is unhealthy for whatever reason, yellowing and browning of the foliage is a typical symptom. In the nursery, yews grown in heavy soils, poorly drained soils and acid soils can lead to above ground symptoms similar to root rot even in the absence of root rotting pathogens. Yews grown in the landscape showing yellowing and browning symptoms may also be subject to transplanting shock, excess soil fill, root injury due to construction activity, dog injury, de-icing salts and excess soil moisture due to accumulated downspout run-off. Laboratory analysis may be needed to diagnose Phytophthora root rot as a specific cause of yew losses.

Control of Phytophthora root rot of yew is accomplished by strict sanitation during propagation and by careful selection of sites for field planting that are well drained and have a history of freedom from Phytophthora. See general section on Phytophthora.

Table 17. Some Diseases of Minor Crops.

R. K. Jones, R. C. Lambe and G. W. Simone

Host Name	Disease	Pathogen	Comments
Abelia grandiflora—abelia	no important diseases		
Abies spp.—fir	damping-off	*Pythium* sp., *Rhizoctonia solani* *Phytophthora cinnamomi*	seedling disease serious on *A. fraseri*
Acer spp.—maple	tar spot anthracnose damping-off wilt	*Rhytisma acerinum* *Gloeosporium apocryptum* *Pythium* sp., *Rhizoctonia solani* *Verticillium albo-atrum*	 early summer, wet weather seedling disease
Ajuga reptans—bugle-weed	southern blight	*Sclerotium rolfsii*	summer disease
Albizia Julibrissin—mimosa	wilt	*Fusarium oxysporum* f. sp. *perniciosum*	cultivar "Union" resistant
Betula spp.—birch	crown gall root knot nematode	*Agrobacterium tumefaciens* *Meloidogyne* spp.	
Callistemon spp.—bottlebrush	leaf spot stem gall	*Cylindrocladium scoparium* *Sphaeropsis tumefaciens*	 serious problem
Carissa spp.	root rot root rot aerial blight, dieback stem gall	*Phytophthora parasitica* *Pythium* spp. *Rhizoctonia solani* *Sphaeropsis tumefaciens*	 serious in deep South
Carya illinoensis—pecan	crown gall scab	*Agrobacterium tumefaciens* *Cladosporium effusum*	serious problem
Cercis canadensis—redbud	damping-off canker	*Pythium* sp., *Rhizoctonia solani* *Botryosphaeria dothidae*	seedling disease damaging
Cleyera japonica—cleyera	anthracnose	*Colletotrichum* spp.	not serious
Cotoneaster spp.—cotoneaster	fire blight web blight	*Erwinia amylovora* *Rhizoctonia solani*	spring, wet summer
Chamaecyparis spp.—white cedar	blight root rot	*Phomopsis juniperovora* *Phytophthora cinnamomi*	
Cupressus sempervirens	twig blight	*Phomopsis juniperovora*	serious on new growth
Daphne odora—daphne	root rot	*Phytophthora cinnamomi*	serious problem
Eleagnus spp.—Russian olive	canker	*Phomopsis* sp.	
Eriobotrya japonica—loquat	leaf spot fire blight	*Entomosporium maculatum* *Erwinia amylovora*	spring, wet spring, wet
Eucalyptus spp.	leaf spot leaf spot	*Cercospora* spp. *Cylindrocladium* spp.	serious problem
Fatshedera spp.	anthracnose root rot root and stem rot	*Colletotrichum* sp. *Phytophthora parasitica* *Rhizoctonia solani*	
Fatsia japonica	root rot	*Pythium* sp.	
Ficus spp.	anthracnose rust	*Colletotrichum gloeosporioides* *Cerotelium fici*	 spring, fall
Forsythia spp.—golden bells	root rot southern blight crown gall	*Phytophthora* sp. *Sclerotium rolfsii* *Agrobacterium tumefaciens*	
Gardenia jasminoides—cape-jasmine	root knot nematode canker root rot leaf spot	*Meloidogyne* spp. *Phomopsis gardeniae* *Rhizoctonia solani* *Mycosphaerella gardeniae*	
Gardenia radicans	root knot nematode	*Meloidogyne* spp.	very susceptible
Hedera helix—English ivy	root rot anthracnose leaf spot	*Pythium* spp., *Phytophthora* spp., *Rhizoctonia solani* *Colletotrichum trichellum* *Xanthomonas hederae*	

Table 17 Continued.

Host Name	Disease	Pathogen	Comments
Hydrangea spp.—hydrangea	powdery mildew	*Erysiphe polygoni*	
	blight	*Botrytis cinerea*	flowers and flower buds
	bacterial wilt	*Pseudomonas solanacearum*	soil-borne pathogen
	leaf spot	*Cercospora* spp.	
	leaf spot	*Corynespora cassiicola*	
Ixora spp.	twig canker, stem gall	*Sphaeropsis* sp.	
Kalmia latifolia—mountain-laurel	leaf spot	*Mycosphaerella colorata*	serious in shade
	root rot	*Phytophthora cinnamomi*	
Liriope spp.—liriope	anthracnose	*Colletotrichum* spp.	on old leaves, little damage
Ligustrum spp.—privet	leaf spot	*Cercospora* spp.	serious in deep South
Magnolia spp.—magnolia	algal leaf spot	*Cephaleuros virescens*	
	leaf spot	*Phyllosticta magnoliae*	
Magnolia soulangeana	bacterial leaf spot	*Pseudomonas syringae*	spring, wet
Mahonia spp.—Oregon grape	leaf spot	*Phyllosticta mahoniana*	
	brown leaf necrosis	*Cylindrocladium ellipticum*	
Myrica cerifera wax-myrtle	no important diseases		
Nandina spp.—heavenly bamboo	mosaic	*Cucumber mosaic virus*	common in dwarf types and
		Tobacco ring spot virus	landscape plants, not seed borne
	root rot	*Pythium* spp.	poor drainage
Nerium oleander—oleander	canker	*Sphaeropsis* spp.	
Osmanthus spp.	no important diseases		
Pachysandra terminalis	root rot	*Pythium* spp.	
	leaf and stem blight	*Volutella pachysandrae*	
Pieris spp.—andromeda	dieback	*Phytophthora* spp.	see rhododendron
	root rot	*Phytophthora* spp.	see azalea
Platanus spp.—sycamore	anthracnose	*Gnomonia platani*	cool wet spring
Prunus spp.—cherry	leaf spot	*Coccomyces hiemalis*	
	root knot nematode	*Meloidogyne* spp.	
Prunus spp.—plum	black knot	*Apiosporina morbosa* (*Dibotryon morbosum*)	
	leaf spot	*Xanthomonas pruni*	
	root knot nematode	*Meloidogyne* spp.	
Prunus persica—peach and almond	brown rot	*Monilinia fructicola*	
	leaf curl	*Taphrina deformans*	
	root knot nematode	*Meloidogyne* spp.	
	crown gall	*Agrobacterium tumefaciens*	
Pyracantha spp.—firethorn	scab	*Fusicladium pyracanthae*	see crab apple
	fire blight	*Erwinia amylovora*	see "general" section
Salix babylonica—willow	crown gall	*Agrobacterium tumefaciens*	
Syringa spp.—lilac	powdery mildew	*Microsphaeria penicillata*	
Ulmus parvifolia—Chinese elm	black leaf spot	*Gnomonia ulmea*	fall, cool
	powdery mildew	*Phyllactinia guttata*	
Viburnum spp.	leaf spot	*Cercospora* spp.	

Introduction to Disease Control

R. K. Jones

The goal of every nurseryman should be to grow disease-free plants. In the preceding chapters, we have discussed the major diseases of woody ornamentals, their causes and the environmental and physical factors affecting their development. This knowledge is a prerequisite for developing effective disease control strategies.

There are many facets to a total disease control program. Control procedures can be simple, complex, integrated, economical or costly. The combination of procedures used will depend on the type of disease and pathogen encountered, how soon it is detected and diagnosed and the number of plants or size of area involved.

Disease control must become a "frame-of-mind". This total control program is first and foremost a management concept. A nursery must be managed and operated to keep pathogens out, to provide a favorable environment for plant growth and to avoid disease occurrence. Disease control after infection has occurred is often not possible or is frequently more expensive in the long run than disease avoidance.

To be effective, disease control in the nursery must be a total, continuous, preventive program, starting before the cutting is inserted in the rooting bed (propagation) and continuing until the finished plant is sold. Control measures such as sanitation, resistance, fungicides, nematicides and biologicals are topics which will be discussed separately in the following chapters.

Sanitation, resistance and cultural control will be strongly stressed as the basis of the total disease control program. Chemical control should be supplementary to and not a substitute for sanitation and cultural control. Specific chemical control measures are not recommended in this publication because they are constantly changing. A small number of chemicals are legally cleared for use on woody ornamentals and the chemical registrations and recommendations may vary from state to state. Therefore, specific chemical recommendations should be obtained locally.

Fungi

Since most diseases of woody ornamental plants in nurseries are caused by fungi, most of the recommended control practices are aimed at fungal diseases. For control programs, diseases caused by fungi can be divided into two general groups: 1) aerial—diseases of stems, leaves and flowers caused by fungi that produce spores that spread in the air or water; and 2) soil-borne—diseases of roots and lower stems.

Aerial diseases are generally easier to control than soil-borne diseases. Aerial diseases are controlled through a combination of sanitation, environmental manipulation, resistance and fungicide sprays. Photinia leaf spot caused by *Entomosporium maculatum* can frequently be controlled by taking cuttings from disease-free stock plants and growing the rooted cuttings isolated from other diseased plants of *Photinia, Raphiolepsis* and loquat. This disease is usually less severe on field grown *Photinia* than those grown in containers that are watered frequently overhead. *Photinia serulata* has been replaced in the nursery trade partly because *Photinia fraseri* is *resistant* to powdery mildew. Fungicide sprays are frequently used to aide in the control of such areial diseases as powdery mildew, blackspot on rose and Photinia leaf spot.

Soil-borne disease control is based heavily on sanitation and soil environmental manipulation. Such practices as surface water control, pathogen-free potting media, pathogen-free liners, careful use of irrigation, well-drained potting media or soil for field plantings have been the backbone of soil-borne disease control programs. Resistance has not been utilized as much as it could be because cultivar resistance evaluations have not been made or have been made only recently and ornamental plants are chosen more on horticultural or landscape characteristics rather than disease resistance. Fungicides have not been widely used nor effective for soil-borne disease control in woody ornamentals because: a) available fungicides act as fungistats that do not kill the pathogen and thus require frequently repeated applications; and b) fungicide drenches require an excessive amount of labor. New fungicides may be more effective for control of Phytophthora root rot than materials available in the past have been. Biological control may be effective with this group of diseases in the future.

Bacteria

Bacterial diseases are not numerous on woody ornamentals but several cause economic losses and are not easy to control under nursery production conditions. Preventive control measures are essential. Excluding bacterial pathogens from a planting involves making use of pathogen-free seeds and plants. Sanitation involves removing and destroying infected plants or plant parts in and around a planting. Decontaminating hands and tools and sterilizing soil helps prevent the spread of the pathogen. Cultural practices such as fertilizing and watering can be adjusted and crop rotation can be used to reduce some bacterial diseases. Use of resistant varieties is very effective in bacterial disease control. Chemical control of bacterial diseases has had limited success. Antibiotics and copper-containing chemicals have been utilized in combination with other practices listed above to aid in control of some bacterial diseases. Biological control of crown gall may indicate future possibilities for control of other bacterial diseases.

Viruses

Virus diseases can be controlled best by eradicating sources of virus. On woody ornamental plants, asexual propagation is the most important method of spreading virus diseases. Mother plants for cuttings and scion wood need to be inspected periodically for symptoms and diseased plants eliminated. Meristem culture has been used on some crops to produce virus-free mother plants. Weeds and various perennial plants around nurseries and greenhouses can be sources of vectors and viruses and should be controlled. Insect vector control at the virus or vector source is much more likely to prevent virus spread than trying to protect ornamental plants with an insecticide. Control of the nematode vector genera (*Xiphinema, Longidorus, Paralongidorus, Trichodorus* and *Paratrichodorus*) by soil fumigation will prevent spread of nematode-transmitted viruses. No chemicals are available to cure a virus infected plant.

Nematodes

In southeastern nurseries, nematodes should be a rare problem on woody ornamentals grown in soilless media in containers. This problem is avoided under these conditions through a good sanitation program since media components, new pots and liners are generally free of nematodes. With plants grown in soil, either in pots or field, nematodes can be a common problem. Control strategies under these conditions include selecting planting sites free of pathogenic nematodes, use of preplant soil fumigation, use of resistant or immune cultivars or species or use of post-plant contact nematicides.

Mollicutes

There are few diseases caused by mollicutes that are important in the woody ornamental nursery industry in the Southeast. Since these pathogens are systemic within the plants, mollicute diseases must be avoided. This is often accomplished through the use of resistant cultivars or immune species. Certain antibiotics such as tetracycline, chloramphenicol and erythromycin will inhibit mollicutes in plants. These chemicals will not cure diseased plants, however, and are not used on a commercial scale. Control of leafhoppers, and weeds which may be sources of leafhoppers and mollicute pathogens, is important.

Disease Management in the Nursery

G. W. Simone

Disease management is an "umbrella" concept that gathers and organizes all existing disease control measures that are available to the nurseryman. Normally, this concept is subdivided into three courses of action:

1. Prevention—activities that occur prior to disease incidence.
2. Monitoring—early detection of disease during the plant production phase.
3. Management—control activities selected to reduce disease damage.

DISEASE PREVENTION

Since most nurseries have distinct areas for stock plants, propagation and production, the phases of disease management will be discussed for each area.

Preventive practices in the nursery include both common sense and an attitude directed toward sanitation. The section on "Sanitation" presents a comprehensive overview of disease preventive practices for the nursery. The following pertain to specific areas in the nursery production cycle and are presented to compliment the "Sanitation" section.

I. *Stock Plant Area (Mother Blocks)*

1. Land for mother block establishment should be cleared. All roots of hardwood species, especially native oaks, should be removed. Residual root debris in soil often serve as a food substrate for the shoestring and mushroom root rot fungi (*Armillariella mellea* and *Armillariella tabescens* respectively).

2. If soil-borne diseases have been a problem in the past (such as bacterial crown gall and root knot nematode injury), apply a broad spectrum, preplant soil fumigant or nematicide.

3. Field grown stock blocks must allow adequate spacing between plants. This spacing will allow rapid drying of foliage, impede the spread of pathogens and other pests among plants and facilitate better spray coverage of pesticides.

4. Application of irrigation water should be so timed to consider the amount of natural rainfall. This will avoid saturated soils or droughty conditions that can stress root systems. Plant stress is one predisposing factor to root diseases.

5. For woody ornamental species with persistent foliar disease problems, irrigation should occur between midmorning to early afternoon; always avoid wetting foliage prior to night hours. Daytime watering has been shown to aid in reducing foliar disease severity for certain species.

6. See "Sanitation" section for more information.

II. *Propagation Area*

1. Propagation cycles of a particular species should be timed to maximize the most rapid rooting for that species. This would limit exposure of cuttings to potential cutting or root rotting organisms.

2. Water sources should always be checked for contamination by root rotting fungi. This should be the case when a root rot problem has been a persistent problem in a particular area for several years. Shallow water systems (such as ponds, rivers, creeks, canals or shallow wells) *should not* be used for irrigation in propagation.

3. In coastal areas where salt water intrusion is a problem, nurserymen should not utilize a water source that is high or variable in soluble salts for propagation. High salts can stress root systems thereby predisposing young liners to root disease.

4. See "Sanitation" section for more information.

III. *Production Area*

1. Woody species that are commonly affected by certain diseases can be grouped in adjacent areas within the nursery. This will allow for a greater ease in scouting and a more rapid detection of diseases. Better pesticide management of diseases is usually the result. Examples of pathogens attacking several crops include *Entomosporium* leaf spots, *Cylindrocladium* aerial blights and powdery mildews.

2. Restrict nursery beds or block sizes to allow for a more efficient pesticide application with existing spray equipment. Block sizes should be based upon the spread of the mature stock, not newly planted liners. Spacing between plants on beds allows for a better spray penetration of plants. This is not practical for 3 to 4 gallon containers.

3. Certain foliar diseases are a yearly problem on the same ornamental species. Consider blocking these disease-susceptible plants together for day watering. Midmorning to early afternoon irrigation will minimize the actual hours the foliage remains wet. This practice reduces the potential for foliar diseases.

4. See "Sanitation" section for more information.

MONITORING

A systematic monitoring system (disease scouts) by nursery personnel aids in forecasting certain seasonal disease problems. The recognition of a particular production situation or practice that predisposes plants to disease would be recognized via the scouting activities, and thus, these problems could be corrected before disease becomes a serious problem. The scouting system is effective only when followed by rapid deployment of management practices. Therefore, a weak link between detection and disease control negates the usefulness of an entire disease management concept.

I. *The Scout*

Within the nursery, an individual could be designated as the "disease scout". This could be the owner, the head grower, a few workers or even all employees. There is no preferred scout model for all nurseries. The selection of a scout in a woody ornamental nursery is determined by many factors which should include nursery size, diversity, number of employees and available expertise and experience. One thing that has been defined is the scout profile—a compilation of attributes that a scout must have if such an individual is to be effective.

To qualify as a scout, an individual must have a horticultural background. Being able to recognize a healthy plant and knowing how to grow a particular plant properly will allow individuals to perceive an unhealthy plant quickly. Perception is probably the second most important attribute of a potential scout. Subtle changes in leaf size, vigor, color or the early stages of a particular disease (such as leaf spots or galls) must be observed quickly if such a scouting program is to be valuable to a nursery. A working knowledge of plant diseases associated with many plant species grown in a commercial nursery is required. Finally, when a disease problem is detected, common sense is needed to effectively evaluate the magnitude and type of action to take.

Very few nurseries have available to them an employee who adequately fits all attributes of the scout profile; however, there are still some things that a grower-owner can do to encourage knowledge and skill developments. He can allow interested employees to pursue additional education through local or state Cooperative Extension Service educational programs on work time. This will indicate an owner's interest in this disease management concept.

The grower-owner can distribute publications that deal with horticulture, diagnosis and control of diseases and insect problems of nursery crops to his employees and he can display educational material and reminders in working areas to reinforce educational efforts of his employees.

The use of an incentive program through salary or fringe benefits may also stimulate a more effective scouting program.

Presently, the adoption of the disease management scout or consultant concept as presented above is not commonplace in most nurseries. The application of this and other disease management practices are both practical and needed.

The production characteristics of an ornamental nursery are conducive to a disease epidemic if early stages of disease development are not detected. Therefore, more efficient and systematic disease monitoring practices should be incorporated into current maintenance activities by expanding the labor force or utilizing the existing one. Therefore, the obvious gains from early disease detection include: a) reduction in plant loss; b) improvement in plant quality; c) reduction in production costs; d) circumvention of production delays; and e) increased profits.

II. *Stock Plant Area*

1. This area should be routinely inspected every 2 to 4 weeks throughout the growing season. Two months prior to taking cuttings, this routine inspection should occur weekly.
2. Inspect a minimum of 5 percent of the total plants in a block of one species and choose different plants each time.
3. Begin inspection of plants from the windward side of a block and/or the side bordering an uncultivated area.
4. Inspect plants in a random fashion; check new and old growth, but concentrate predominantly on the new, more susceptible growth. Examine both leaf surfaces for disease symptoms. Observe leaves for a decrease in size, change in color or general loss in vigor. These symptoms could indicate lower stem or root problems.
5. When common diseases are detected in a nursery, all susceptible plant species should be carefully monitored on a regular schedule. The incidences of diseases that are common to a number of woody ornamentals will depend upon a specific nursery's geographical location in the southeastern United States. Some examples of common diseases which occur on several different hosts over a wide geographical area are listed in Table 18.

Table 18. Common Diseases Occurring on Several Different Hosts.

Cylindrocladium	*Entomosporium*
Azalea	Cotoneaster
Callistemon	Crab apple
Eucalyptus	Hawthorn
Feijoa	Loquat
Magnolia	Photinia
Rhododendron	Quince
Leucothoe	Raphiolepis
	Apple
	Pear
Fire Blight	*Rhizoctonia Web Blight*
Cotoneaster	Azalea
Loquat	Carissa
Mountain Ash	Euonymus
Pyracantha	Hibiscus
Spirea	Ilex
Apple	Juniper
Pear	Viburnum
Photinia	Cotoneaster

6. Record where diseases occur and their precise location in stock beds. This could help in the planning of future monitoring and management activities.

7. Disease symptoms alone are often unreliable in a diagnosis; therefore, seek professional diagnostic services. Contact your local Agricultural Extension Service for instructions on the proper collection and submission of disease samples to a plant disease diagnostic laboratory.

III. *Propagation Area*

1. Propagation beds should be routinely inspected on a 1 to 2 day interval while plants remain in this cycle.

2. Be sure misting systems function properly; examine rooting media for water saturation. This "wet feet" condition has been shown to predispose seedlings or cuttings to diseases.

3. Examine seedlings for damping-off disease. Damping-off appears as a small number of plants within a small area that have wilted or collapsed at the soil line.

4. Propagation beds should be inspected for localized "hot spots" of root or cutting-end rot diseases. Foliage of cuttings will darken and drop from plants exhibiting obvious cutting-end discoloration or rot.

5. Be aware of leaf diseases which often occur on foliage of cuttings. The propagation environment is ideal for disease spread.

6. Collect and submit appropriate samples to the nearest diagnostic facility for an accurate diagnosis of any questionable disease problem.

IV. *Production Area*

1. Plant specimens purchased from outside the nursery should be placed in a holding or quarantine area for at least 3 weeks. Make weekly inspections for insects and diseases so that problems can either be corrected or plants discarded.

2. Production areas should be monitored, especially where diseases are more likely to develop; this is of significant value in preventing diseases. Many production activities can predispose plants to root or foliar pathogens. The following examples illustrate some disease inducing situations that are often observed in nurseries during routine scouting:

a) Puddling water on beds after irrigation that remains for extended periods can predispose plants to root diseases.

b) Standing water along the front edge of beds or blocks shows a need for drainage tiles or ditches. Root disease is often associated with this type of situation.

c) A zone or band of off-color plants across a block may or may not be exhibiting wilt or dieback symptoms. The cause of such problems has been associated with overlapping irrigation patterns or two irrigation zones that fail to meet.

d) Torn, ground plastic on beds allows movement of soil pathogens through container drainage holes into container media and results in root rot disease.

e) Accumulated plant debris from "housekeeping" activities or from pruning or shearing operations that remain in the immediate vicinity of production beds provide excellent "food" for pathogens. This leads to the development and/or persistence of certain plant diseases.

f) Close proximity of dead plant disposal areas with propagation areas greatly increases the continued dissemination of pathogens back into the production cycle.

g) The practice of re-setting rooted cuttings or liners into containers located in the field where plants have failed to grow could be detrimental to the plant. Soil pathogens, if not responsible for the first plant's death, may exist in the plant residue in increased populations.

3. Production beds for diseases should be inspected every 10 to 14 days; this should involve a minimum of 5 percent of the total plants per bed. Select different plants for each subsequent inspection.

4. Inspect both the windward side and/or any side bordering an uncultivated area; foliar pathogens can be expected to enter a production site from either area.

5. Monitor randomly in blocks. Check both old and new growth for obvious disease symptoms.

6. Be alert for root rot symptoms during monitoring. Roots of plants exhibiting decreased vigor, off-color, smaller leaf size or wilt should be examined. Remove the container and examine the root system. Peripheral roots that are dark, discolored and sluff easily to the touch indicate root problems. Lack of root penetration in the lower one-third to one-half of the potting media is another indication of root problems.

7. When a particular disease is detected on several plant species, all other known susceptible species should be carefully examined.

8. Record all disease observations and associated production problems for future monitoring and management activities.

9. When specific diseases cannot be accurately diagnosed on site, collect and submit appropriate samples to a plant disease diagnostic laboratory in your state.

MANAGEMENT

Nurseries that have used disease management programs indicate that the difficulty lies in the management phase rather than in the disease detection components. Nurseries that use scouts, employees or special consultants obtain adequate disease detection, but translation into disease management activities is often lost or ignored. Growers understand the need for active control measures, but these actions are often given a low priority within the nursery as compared to the propagation, potting, irrigation and sales. The old adage of "better late than never", can prove to be disastrous. Therefore, nursery owners and managers need to assign higher priorities to actual disease control activities in the future.

Developing specialization within existing nursery labor forces to control potential diseases can be beneficial in making management decisions. Individuals within the nursery could be given the responsibility for receiving disease monitoring reports on which disease management decisions are made. A group within the nursery could be responsible for all disease control activities including pesticide application. The departmentalization of a pest control group would help to develop a greater expertise in the selection and application of pesticides. Increased consistency in technology and safety as they relate to pesticide application would also result.

After the execution of certain management decisions and actions, an evaluation of such decisions would be beneficial. This would include success of disease control and the practicality of the precise actions taken as they relate to the accuracy of the monitoring system. Such evaluations should be based on visual observations, past records and effects of those control measures used previously. This type of assessment allows for a more positive approach to the preventive phase of disease control and may indicate the need for additional monitoring efforts.

I. *Stock Plant Area*

1. Stock plants with localized disease problems can often be controlled easily and rapidly by selective pruning or handpicking of diseased foliage. A thorough "cleanup" of all infected plants and uninfected plant debris should follow.
2. Certain stock plants with severe disease problems (such as crown gall disease or mushroom root rot) should be rogued from the stock plant area.
3. Obtain the most current and effective control measures from the nearest Cooperative Extension Service office after making accurate diagnoses.
4. Control measures for foliar diseases should be initiated rapidly to insure that cuttings used in propagation are apparently disease-free. As stated previously, the propagation cycle creates a perfect environment for disease epidemics if pathogens are present in plant tissue.

II. *Propagation Area*

1. Areas of declining or dead plants with root diseases within propagation beds should be *carefully* removed—rooting medium and all. Certain root-rotting fungi (such as *Sclerotium rolfsii* or *Rhizoctonia solani*) will produce an asexual, reproductive structure called a "sclerotium." These structures are quite small and can be moved by various means from one site in the propagation bed to another. Extreme care should be taken when roguing individually infected plants.
2. Where a root rot is the problem, apply an appropriate drench fungicide in the propagation bed. Drench the *entire* bed, not just "hot" spots. Since some soil fungicides are specific for a particular organism, an accurate diagnosis is imperative. Consult your Cooperative Extension Service for recommendations. Root rot incidences should always be recorded. Rooting media should never be reused.
3. Severe root rot diseases of plants in propagation beds do not affect all plants; some do survive. It is always tempting to use these surviving plants which should be discarded. Since most growers will pot survivors, be sure to treat the potting mix with an appropriate fungicide as a protective measure. Monitor plants, plus repeat fungicide application at intervals described on the label.
4. Rooted liners exhibiting foliar disease symptoms should be sprayed with an appropriate fungicide before introduction into the production area; continue on a regular basis. Consult your local county Extension agent for specific recommendations.

III. *Production Area*

1. All fungicide selection and application must be based on the latest and most accurate recommendations.
2. Some diseases occurring on aerial portions of plants can be controlled by selective shearing or pruning of infected plant parts. Removal of severely blighted leaves will improve the efficacy of subsequent fungicide applications.
3. Severely diseased plants should be rogued from the production site and appropriately discarded. Plants that must be rogued specifically are those with root rot, crown gall and southern blight. This is especially important since chemical control for these diseases is unsatisfactory.
4. Disease management decisions directed toward root rot disease control should consider these factors:
   a) Root rot diseases caused by *Phytophthora* and *Pythium* spp. are prevalent; disease development is favored by saturated soil moisture levels. Frequent and/or excessive irrigation will favor the continued reproduction and spread of these fungal pathogens. Therefore, proper water management must be recognized as an important facet of disease management.
   b) When root diseases exist, avoid water or fertility extremes. Both will place additional stress on an already weakened plant.
   c) Systemic fungicides are more effective than protectant fungicides. These compounds affect the target fungus in plant tissue.
   d) Drench fungicides applied to soil to control root rotting fungi should be deposited on and in the potting media and not applied to foliage. If these products are applied through overhead irrigation, they should be washed-off foliage by additional water.
   e) Moisture content of potting soil should be at or near container capacity when a fungicide drench is applied. Avoid applications to very dry mixes; this can cause severe root damage.

5. For foliar situations, certain facets of the disease cycle should be reviewed prior to control activities.

a) Most foliar fungal pathogens require long periods of leaf wetness if infection is to take place. This is one reason that late afternoon rainfall or irrigation is not good. When foliar disease severity is high, minimize the irrigation cycle.

b) Application of fungicides or bactericides should *not* be expected to control a disease immediately. The variable length of time between penetration by a spore or bacterium and the appearance of the first recognizable symptom of disease (such as leaf spot) is the incubation period. This time may span days or weeks and will continue in spite of the application of a protectant fungicide with no kickback action onto the plant surface. This means that the disease can be expected to increase in incidence and severity until all incubation periods that began before pesticide applications, become recognizable symptoms. Do not give up on a pesticide prematurely.

c) Avoid foliar pesticide application to wilted plants.

d) Many diseases necessitate pesticide coverage on both leaf surfaces to be effective. Leaf underside coverage is particularly important with certain fungal pathogens like the *Cercospora* spp. on ligustrum and pittosporum that reproduce from the lower leaf surface.

e) Irrigate plants prior to spraying to allow the longest contact of pesticide and foliage prior to the next watering.

Sanitation: Plant Health from Start to Finish

Eugene H. Moody, Sr., and Gerald E. Smith

The key to the financial success of most southern ornamental nurseries is to increase production and at the same time reduce costs. The only viable solution to reducing costs in this rapidly inflating economy is to minimize plant loss and maximize plant quality. If nurseries are to survive economically, production efficiency must be emphasized more now than in the past.

Marketing plants and disease losses are two of the most important factors in ornamental production. Root rots and stem diseases are economically the most important disease groups because of control difficulties. Foliage and flower diseases are usually more convenient to control simply because they occur above ground. The objective of this section is to describe a plan of action useful to nurseries in reducing disease losses. In order to succeed, it is essential that disease control measures be incorporated into every facet of the production cycle.

It is difficult to predict future developments and changes in nursery production; therefore, each grower must develop a sanitation program to fit a particular situation. Before a disease control program can be successfully established, growers must understand the scientific principles that form the foundation of the most recommended and successful practices.

The nurseryman should understand that organisms which cause disease, especially those of the roots and stems, make their way into the nursery production cycle because certain safeguards are not built into the system. One way to reduce introduction of disease-causing organisms into the nursery production cycle is to define and understand the potential sources. Some of the most common sources of contamination associated with nursery production are as follows: 1) contaminated soil splashed about by drops of water from irrigation or rain; 2) pathogens deposited on cuttings which are placed in contaminated water or hormone solutions; 3) hoses dropped carelessly to the ground (pathogens get into the nozzle-end and are expelled into pots or on benches at the next watering); 4) pathogen infested soil and organic material not removed from used flats, pots, benches or other containers between plantings; 5) contaminated soil carried on tools, covers, or worker's hands; 6) infested soil deposited on sterile potting mix, disinfected benches or flats by foot traffic; 7) such things as flats or plants placed on the ground; and 8) planting infected seed, cuttings or seedlings. Because there are so many sources of contamination, emphasis should be placed on practices which keep the pathogens out (exclusion) of the production cycle.

A common approach to disease control in many nurseries is to correct one specific problem at a time; whereas, in actuality, these problems must be solved by making changes in the entire production cycle and not by the usual piecemeal approach. Sanitation is probably one of the most important functions in a nursery and must be viewed as a series of different functions within the nursery production cycle which is analogous to the links in a chain; one broken link can result in financial disaster through losses due to plant disease.

Disease control in plant production must, therefore, begin with the seed or stock plant and end with the marketable product.

Developing the Sanitation Program in Propagation

Plant disease development in the nursery is very seldom an isolated incidence. Disease control should begin with propagation.

I. Establish Pathogen-free Stock Plants

Certain safeguards must be taken to assure that those stock plants from which cuttings are taken remain healthy. Consider the following points in maintaining healthy mother or stock blocks:

1. Isolate all new stock plants to determine their health status prior to introduction into the existing nursery.
2. Stock plants or mother blocks should be isolated from all possible sources of infection. Weedy and known disease areas should be avoided.
3. Establish an independence between the production and merchandising part of the nursery and the maintenance of the mother blocks of stock plants. Mother blocks have been effective because of the ease and the low cost necessary to maintain such a small area.
4. Practice regular spray schedules to control foliage, stem or flower diseases. Drenching with soil fungicides is added insurance. Check with your state Cooperative Extension Service for specific chemical recommendations in your area.
5. Always follow appropriate sanitary practices and those applicable considerations described above and elsewhere in this publication in the proper maintenance of mother blocks.

II. Collection of Cuttings

Success in propagation is often realized by observing reasonable sanitary procedures when collecting cuttings. Some of the more prevalent procedures are described below.

1. Collect cuttings from tops of healthy plants. Top cuttings are usually free of soil-borne pathogens. Plants grown on above-ground level supports or trellises are generally free of soil pathogens.
2. Avoid taking cuttings from plants at or near the soil level.
3. Never use root divisions unless absolutely necessary.
4. It is better to break rather than cut cuttings from plants.
5. If knives or pruning shears are used to collect cuttings, have each worker use two separate pairs—one left soaking in disinfectant while the other is being used. As a suggestion, a household bleach (such as Clorox or Purex) solution (1 part bleach to 9 parts water, 1:9) or 70 percent alcohol is effective (see Table 19). Keep in mind that the bleach solution will cause rusting of ferrous metals. Therefore, thoroughly clean cutting blades twice daily. Frequent sharpening is also a must. (Note: Bleach solutions should be changed every 30 minutes since the disinfecting qualities diminish rapidly.)
6. Place cuttings on chemically disinfected surfaces of benches, flats and baskets. A chlorine bleach solution (1:9) is a good surface disinfectant. Mix up new solutions approximately every 30 minutes. Occasional use of copper naphthenate (2 percent) will disinfect wooden surfaces. Some growers spread unused newspaper or wrapping paper on disinfected work surfaces for added insurance. Never place cuttings on the ground.
7. Never dip cuttings in water unless the water is pathogen-free. This is a disadvantage of the liquid rooting hormone formulations.
8. Some growers dip or soak cuttings in a fungicide suspension prior to sticking. Change the solution as often as is economically possible to maintain disinfecting concentrations.

III. Preparation of Flats to be Used in Propagation

Flats should be disinfected either after propagation or before reuse.

1. Thoroughly remove rooting media adhering to the surfaces of flats or other containers with a brush before placing in a disinfectant (1:9 chlorine bleach or 2 percent copper naphthenate).
2. Galvanized metal trays are preferred over wood because they are easier to clean and disinfect (see Table 19).

Table 19. Treatments for Tools, Equipment, Pots and Flats

	Rate to Use		Relative Effectiveness					
Material	**Formulated**	**Application**	**Weeds**	**Nematodes**	**Insects**	**Bacteria**	**Fungi**	**Use, Remarks**
alcohol (grain, rubbing, wood) (70-100%)	Full strength	Dip or swab; do not rinse	poor	fair	poor	good	good	Items that are being treated should be clean and moist and temperatures above 60°
formaldehyde (37%)	1.0 pt/12 gal	Dip or swab	poor	fair	poor	good	good	
sodium hypochlorite 5.25% (Clorox)	10 gal/100 gal	Dip 1-10 seconds, brush, spray, let drain, do not rinse	poor	poor	poor	good	good	
methyl bromide	3 to 11 lb/100 cu ft	Cover items under airtight plastic. Release fumigant in dish.	good	good	good	good	good	
steam	Heat object 180-200°F for 30 min	Cover or otherwise contain steam or heat around object	good	good	good	good	good	Excellent for permanent installation
dry heat			good	good	good	good	good	Good for small non-plastic objects. May use kitchen oven
Copper naphthenate	2%	Dip or swab	good	good	good	good	good	For wood such as flats or benches

IV. Propagating Media

1. *Never* reuse propagating media.
2. Media should be loose, porous and well-aerated. If propagation mixes contain soil, or if ground beds are used, pretreat before use with methyl bromide or heat.
3. Media must be stored so that it does not become contaminated with disease causing organisms.
4. Propagation media should be mixed on a clean concrete slab so that run-off water (rain or irrigation) will not introduce pathogens. Remember that a clean medium can become contaminated if carelessly handled or subjected to foot traffic.

V. Clean Propagating Areas

1. All benches must be free of infested media, leaves and other refuse.
2. Use a household bleach or commercial disinfectant (see Table 19).
3. Use wooden benches treated with copper napthenate (2 percent). A waiting period is necessary after treatment. Benches treated with most other disinfectants do not require a waiting period (see Table 19).
4. All infested plant debris should be removed from the entire propagation area. Although this is sometimes tedious, it is very effective in reducing disease problems. Some growers provide their workers with carpenter aprons in which to place this debris during daily activities. This discourages dropping the plant debris on the ground. A *covered* garbage can should be placed at the end of each greenhouse for plant debris. Cans without lids have little value in a sanitation program.
5. Apply regular fungicide spray schedules to aid in the control of diseases which occur above ground. Chemicals which are cleared for nursery use vary in each state; therefore, contact your state's ornamental plant pathologist for the proper recommendations.
6. As added insurance against contamination, rooting media may be *drenched* immediately before or after sticking and at 4-week intervals with a fungicide combination developed to control the water molds, *Rhizoctonia* spp. and other fungi. There are some difficulties with this practice, plus fungicide and dosage rates vary from state to state; therefore, contact your state's Extension service for more details.
7. Avoid unnecessary handling of (clean) media in the propagating containers or beds. Avoid nervously dipping hands into bench media while conversing. Avoid feeling the media unnecessarily for moisture content.
8. Use low water pressure when irrigating to avoid splashing.
9. Prohibit anyone from walking over treated areas.
10. Hose nozzles should be hung on conveniently located hooks.
11. Workers' hands should be washed after working with raw contaminated media, soil or "unclean"plants to avoid introduction of pathogens.
12. Space cuttings properly when sticking. Poor air circulation contributes to an environment that favors diseases caused by *Rhizoctonia* spp.
13. Water source: *Do Not Use Untreated Pond Water in Propagation.* Well water is usually pathogen-free.
14. Avoid excessive overhead irrigation. *Overwatering* is the major culprit in creating the environment for propagation diseases. Many growers are manipulating their misting schedule to allow no mist on plants after 5:00 P.M. On rainy, humid days, misting is terminated earlier in the day. Wet foliage and high moisture regimes in rooting mixes provide a favorable environment for certain serious diseases.
15. Remove any cuttings from containers or even whole flats of cuttings from the propagation area that exhibit disease symptoms; then treat the propagation area with a fungicide or disinfectant. Have a pathologist diagnose the problem so that appropriate action can be taken.

VI. Raised Benches

Benches raised above the ground are highly recommended for propagation.

1. Steam or fumigate benches after each crop. Chemical fumigants should only be used in an empty greenhouse.
2. Be sure raised benches have been cleaned of all soil. Disinfect with household bleach or a commercial disinfectant prior to reuse if fumigants are not appropriate.
3. Replace propagation media between crops; do not reuse.

VII. Ground Beds

Poorly graded areas surrounding ground beds are easily contaminated by normal run-off and are difficult to keep clean. Pathogens in water droplets are splattered about during irrigation or rain and are easily carried into ground beds from contaminated areas. Follow these procedures if ground beds are used in propagation:

1. Thoroughly prepare the soil. Incorporate pine bark to improve drainage. Incorporate fertilizer and lime as needed.
2. Treat with a wide-spectrum fumigant. See your state Extension service for specific information. Always fumigate between crops.
3. Treat walkways as well as propagation beds.
4. Use boards or cinder blocks to build up bed perimeters to avoid flooding.

Developing a Sanitation Program for Liner Production

The basic concepts described earlier in the propagation program are applicable here. Liners should be grown above ground level to prevent contamination by soil-borne pathogens. Special attention should be given to irrigation frequencies and splashing. Keep hose nozzles off the ground and clean and disinfect benches prior to use.

If you must purchase liners, it is important that you obtain high quality, disease-free plants. Numerous growers in the past have purchased diseased liners; this is a losing proposition because the disease probably cannot be cured. Also, the diseased liners introduce pathogens into the nursery.

Developing a Sanitation Program for Plants After Potting

Consider the "common sense" approach described previously. Nursery operations differ here from the propagation and liner cycles. Those things to be considered are as follows:

I. Preparation of Potting Mix

Use a cement slab of the proper elevation to prevent contamination by surface water run-off from surrounding areas. Mixing on-ground often leads to plant failure because of contamination by pathogens, insects and weeds. Ground equipment should be assigned to this area. Ideally, it should not be moved off the slab. Avoid unnecessary foot traffic on the area. Consideration of size should also be made. Many slabs are too small.

II. Potting Mixes

Determine components of the medium to be used. This determines drainage, aeration and water-holding capacities. These factors influence root growth (good or bad). *Too much or too little of the above media characteristics can provide those necessary environmental factors within the mix for disease development.* A well-aerated and well-drained mix is the most ideal.

III. Planting Depth of Liners

Plant no deeper in containers than the liners originally grew.

IV. Storage of Containers and Potting Media

Store on a clean, non-soil surface which is not subject to splashing water or run-off water. A large concrete pad is excellent for media preparation and storage. It is very important to prevent introduction of pathogens during storage and handling.

V. The Container Growing Area

All sanitation program efforts made previously are for naught if plants are placed directly on the ground. Contaminated soil is often splashed into containers and this often leads to disease development. Therefore, do the following:

1. Place containers on plastic sheeting. Prevent "low spots" by constructing beds as smooth as possible. Beds should be crowned by elevating centers of the beds higher than the edges (6 inch drop per 25 feet).
2. Provide drainage ditches at bed edges.
3. Compact the soil prior to spreading plastic sheeting to prevent settling of soil.

Note: Some nurserymen are using gravel on top of the plastic sheeting. Beds constructed in this manner have been shown to reduce contamination by run-off water and to reduce splattering of soil. This approach aids in disease suppression; therefore, there is a corresponding reduction in plant disease losses. Increased cost initially is the main drawback for gravel beds. However, this is offset by increases in the number of saleable plants. There is also an increase in longevity of the container area which reduces the per year labor and material expenditure.

VI. Pond Water—A Potential Threat

It has been demonstrated that many containers are being infested with pathogens coming from contaminated pond water. It is best not to use pond water. If pond water must be used, do not allow run-off water from growing areas to drain into the irrigation pond. If the only source of water is pathogen infested and root diseases are at epidemic proportions, the use of a well-aerated media and fungicide drenches are important. Root-rotting fungi (such as the water molds Pythium spp. and Phytophthora spp.) can exist in an aerated, well-drained media. Infections of the roots by these two organisms are reduced in well-drained media.

VII. Grouping Plants

Different varieties and sizes of plants differ in water requirements. Therefore, it is imperative that plants be grouped according to variety and size in order to establish a proper water management program. Grouping plants for watering will reduce losses due to overwatering and diseases. Proper watering also stimulates plant growth which will result in a faster production cycle.

Examine Roots

A good nurseryman frequently knocks plants out of the container to observe moisture and root condition. Diseases detected early can more easily be controlled. This practice can also reduce pathogen movement to large numbers of plants. Diseased plants should be removed from the growing area as soon as possible. The disposal area should be located well away from the growing area, storage area, potting area and water source.

Figure 41. Failure to follow a strict, total disease control program can cause severe disease outbreaks that result in excessive losses (Univ. of Georgia).

Figure 42. Disease-free stock plants used for cuttings should be maintained in isolated blocks and should receive special care (Univ. of Georgia).

Figure 43. Cuttings taken from the top of stock plants are less likely to be contaminated. Splashing water easily spreads pathogens from the soil onto lower leaves and stems (Univ. of Georgia).

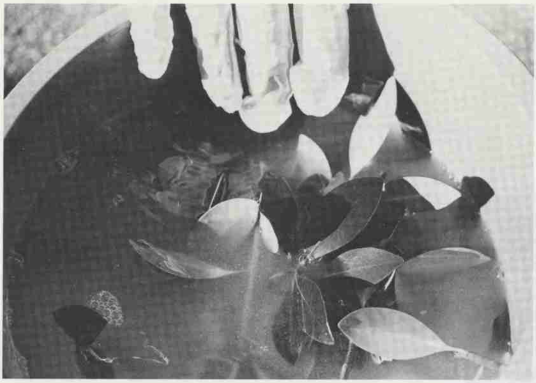

Figure 44. Cuttings of some plants can be soaked in fungicides to reduce certain root rot diseases. Fungicide selection and rate are critical and may vary with each species of plant. Such fungicide solutions or hormone solutions should be changed frequently to minimize contamination (Univ. of Georgia).

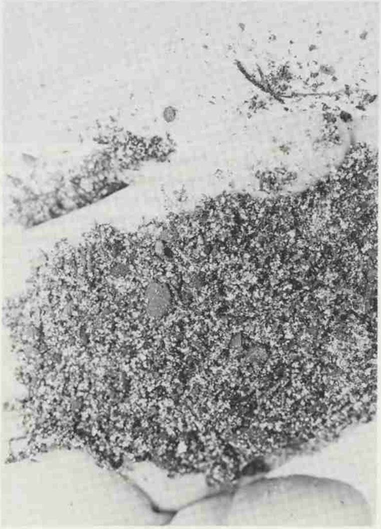

Figure 45. Rooting media *must* be free of pathogens, should drain well and should not be reused (Univ. of Georgia Extension).

Figure 46. If flats are to be reused, they should be thoroughly cleaned and then dipped in a disinfecting solution, such as 2 percent copper naphthenate, chlorine bleach, or other disinfectants, or fumigated (Univ. of Georgia Extension).

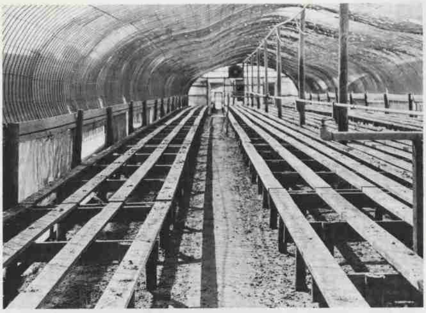

Figure 47. Rooting flats or liners should be placed on pathogen-free benches. After thoroughly cleaning all contaminated soil from benches, treat with a 2 percent copper naphthenate or a 1 to 9 chlorine bleach solution. The greenhouse should contain no plants when either of these chemicals are used (Univ. of Georgia Extension).

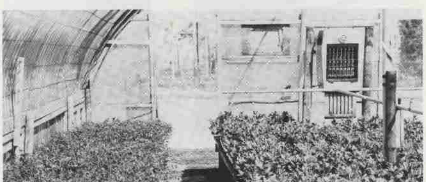

Figure 48. Raised beds are highly recommended for the propagation area. They greatly reduce the possibility of contamination from the untreated soil in the aisles and under the benches (Univ. of Georgia Extension).

Figure 49. Ground beds are less desirable because they are usually poorly drained and are also easily contaminated. Contaminated soil on workers' feet, splashing soil during watering and movement of surface water from surrounding areas are sources of possible contamination (Univ. of Georgia Extension).

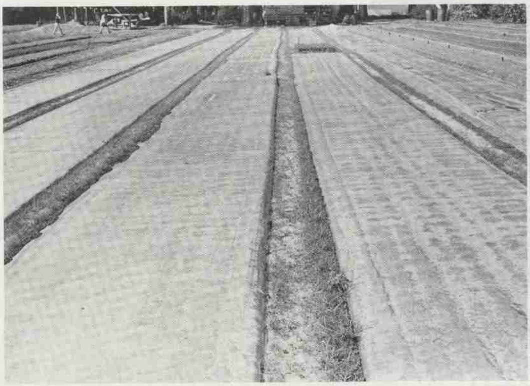

Figure 50. If ground beds are used, they should be treated with a broad-spectrum soil fumigant after thorough soil preparation. The area should be treated before each crop. The situation shown above could be improved by fumigating the walkways (Univ. of Georgia Extension).

Figure 52. As added protection against diseases, fungicides may be sprayed on rooting cuttings if diseases have been a problem during this stage in the past (Univ. of Georgia Extension).

Figure 51. As added protection against root rot disease development, rooting media can be drenched with appropriate fungicides if diseases have been a problem in the past during this stage (Univ. of Georgia Extension).

Figure 53. Liner production should also be off the ground to prevent contamination with pathogens (Univ. of Georgia Extension).

Figure 54. Garbage cans *with lids* should be readily available for disposal of plant debris which must not be thrown on the ground, in walkways or beneath benches (NCSU Extension).

Figure 55. Most of the pathogens that affect cuttings and liners occur naturally in most soils. When the hose is dropped on the ground, the nozzle may pick up these fungi and be spread to many plants during watering (Univ. of Georgia Extension).

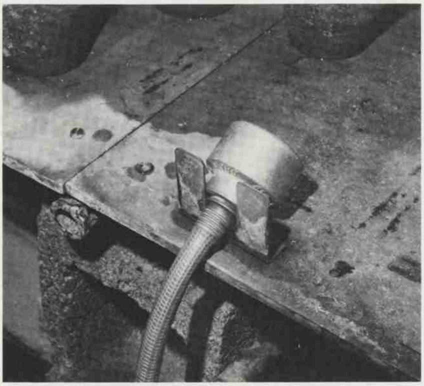

Figure 56. Many different types of simple and inexpensive devices are available or can be constructed to keep hose nozzles off the ground. This practice prevents contamination by pathogens (NCSU Extension).

Figure 57. Mixing and storing media components on the ground is inviting contamination with pathogens and weed seeds (Univ. of Georgia Extension).

Figure 58. Media storage and preparation area is critical in the prevention of contamination by pathogens. A slightly raised concrete pad is ideal (R. K. Jones, NCSU).

Figure 59. The location of the potting media storage and mixing area should be a high, dry area to reduce contamination by run-off water from the growing area (R. K. Jones, NCSU).

Figure 60. Reusing container mixes is inviting disease. Diseases may have been the reason that these containers were dumped. A small amount of contaminated mix can infest a large volume of potting mix. Contaminated mix should be promptly dumped away from the nursery production area (Univ. of Georgia Extension).

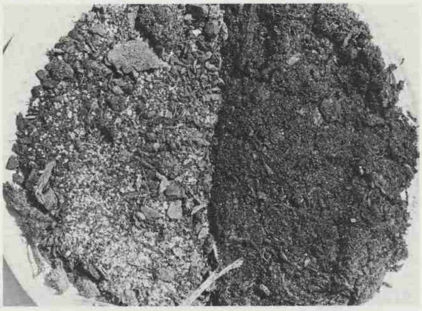

Figure 61. The components of the potting mix will determine the drainage, aeration and water holding capacity of the mix. These factors in turn will affect both plant root growth and root rot development (Univ. of Georgia Extension).

Figure 62. A well-aerated soil mix should lead to the development of an extensive root system throughout the container media (Univ. of Georgia Extension).

Figure 63. This heavy mix held too much water 24 hours after the last watering. This excess soil water will cause root problems by decreasing aeration and providing an ideal environment for the growth and reproduction of several common root rot pathogens (Univ. of Georgia Extension).

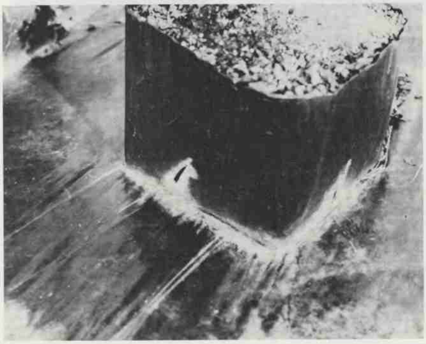

Figure 64. Containers must also have adequate drainage. This type of metal container is very poorly drained and this will keep the media saturated with water, increasing the incidence of root rot (R. K. Jones, NCSU).

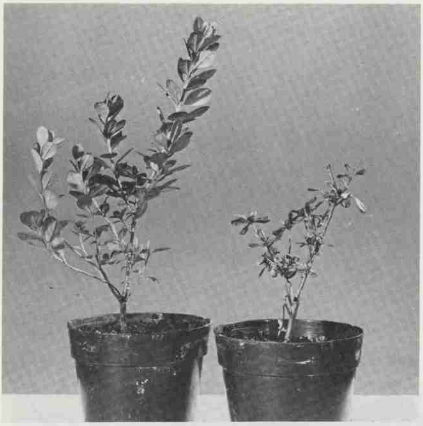

Figure 65. Growth of plants is much faster in mixes that are well-aerated, properly fertilized and pathogen-free. The more vigorous plant on the left grew in a well-drained and pathogen-free mix. Root rot diseases can stunt and weaken plants without killing them (Univ. of Georgia Extension).

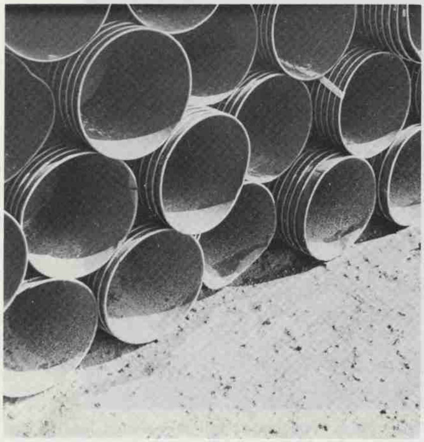

Figure 67. Containers stored on the ground can be contaminated with pathogens in surface water. It is advisable to store new containers in a building or on a concrete pad (Univ. of Georgia Extension).

Figure 66. Setting the liner too deep can adversely affect root development and encourage root rot diseases. Deep planting problems are accentuated by poorly-drained mixes and overwatering (Univ. of Georgia Extension).

Figure 68. A healthy liner in a clean pot containing a well-drained, pathogen-free media is on its way to producing a profitable crop (Univ. of Georgia Extension).

Figure 69. The sanitation program can be broken when containers are set on the ground. Splashing water has contaminated these healthy plants with pathogens (Univ. of Georgia Extension).

Figure 70. Most nurserymen place plastic under containers for weed control. This will also prevent contamination by splashing soil. However, if there are low areas where surface water accumulates, disease development is favored (Univ. of Georgia Extension).

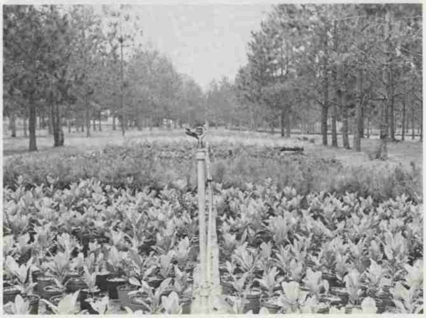

Figure 71. To prevent accumulation of surface water, container areas should be sloped or crowned. The surface should also be compacted to prevent surface pockets that trap water (Univ. of Georgia Extension).

Figure 73. Many nursery irrigation ponds receive surface run-off water from the production area. This run-off water may contain root rotting pathogens which have been shown to survive in ponds and thus can be distributed to all plants in the nursery during irrigation (Univ. of Georgia Extension).

Figure 72. Setting containers on gravel provides excellent drainage of excess water away from the bottom of the container and prevents movement of Phytophthora from container to container (R. K. Jones, NCSU).

Figure 74. The plants in the foreground have different water requirements than those in the background. If proper water management is to be achieved it is imperative that plants be grouped according to water requirements (Univ. of Georgia).

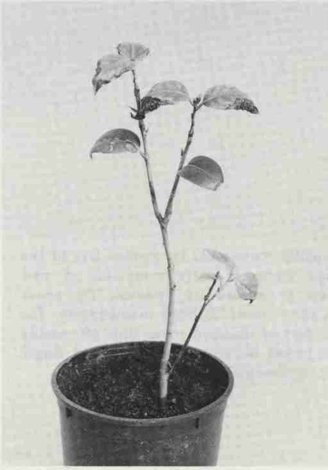

Figure 76. This plant illustrates some symptoms that often occur when roots are damaged or diseased. In addition to marginal leaf burn and leaf drop, new leaves are often chlorotic (yellow) and growth is reduced (Univ. of Georgia).

Figure 75. Plants should be knocked out to observe soil moisture and root condition. Early detection of developing problems may prevent severe damage (Univ. of Georgia).

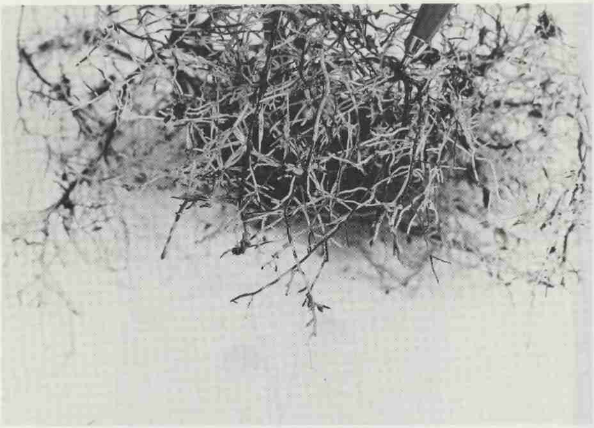

Figure 77. Frequently roots must be washed free of the potting media if early symptoms of root damage or disease are detected. The above roots show brown to black discoloration. The outer part of diseased roots slips off readily when pulled between the thumb and finger (Univ. of Georgia).

Figure 78. Reducing diseases takes additional effort and good management. Frequent use of fungicides cannot make up for the lack of a complete sanitation program. The program will pay for itself through rapid growth, plus more high quality plants to sell (Univ. of Georgia).

Fungicides for Nursery Ornamentals

Larry D. Smith

Modern pesticides reduce the labor-intense tasks associated with crop production and permit individual growers to manage large areas of land. The use of fungicides, insecticides and herbicides in agriculture has increased dramatically in the past 20 years. Escalating costs of labor, land and materials have been largely responsible for the increase. Nurserymen have not escaped this trend and rely heavily on agricultural chemicals to make their operations productive.

Diseases are a major factor limiting nursery productivity. Fungi cause more diseases and produce more severe losses in nurseries than bacteria, viruses, nematodes or mycoplasms. For this reason, chemical control measures often focus on the use of fungicides. This chapter addresses the role of fungicides in nursery production.

Fungicides

Some understanding of the classification of fungi is important to nurserymen because the type of fungus causing a disease will determine the selection of the fungicide. Diseases caused by members of the different classes of fungi often require particular fungicides. Truban®, for example, is effective against the Phycomycetes but will not control members of the Ascomycetes, Deuteromycetes (Imperfects) or the Basidiomycetes. The selectivity of fungicidal compounds is a primary reason that nurserymen must correctly identify a disease and its pathogen prior to selecting a fungicide.

Fungicides are chemicals that kill fungi. Fungistats are chemicals that inhibit but do not kill fungi. Many products are sold as fungicides but act as fungistats under nursery conditions. Broad spectrum fungicides are toxic to many fungal species and are used to control a variety of diseases. Narrow spectrum fungicides are toxic to a small group of specific fungi and are limited in their applications. Narrow spectrum fungicides often provide spectacular control of a few fungi, no control of many fungi, and in some situations may stimulate other fungi.

All fungicides have at least three names: a chemical name referring to the active ingredient; a common name referring to the active ingredient; and one or more trade or brand names. Benlate® is a trade name of a fungicide known to many nurserymen. The chemical name of its active ingredient is methyl 1-(butylcarbamoyl)-2-benzimidazolecarbamate and the common name is benomyl. Benlate® and Tersan 1991® produced by E. I. DuPont de Nemours & Co., Inc. contain benomyl as the active ingredient. Table 20 lists a number of fungicides with their common names and trade names.

Table 20. Common Names and Trade Names of Fungicides

Common Name	Trade Names*	Action**
Benomyl	Benlate	S
	Tersan 1991	S
Bordeau mixture	Many names	P
Captafol	Difolatan 4F	P,E
Captan	Captan	P,E
	Othocide	P,E
	Merpan	P,E
Carbendazim, BCM	Bavistin	S
	Derosal	S
CGA 38140	Fongarid	S,P
Chlorothalonil	Bravo	P
	Daconil 2787	P
	Exotherm Termil	P
Copper	Many names	P
Cycloheximide	Acti-dione PM	P
	Antispray	P
Dichlone	Phygon	P
	Quintar	P
Dicloran, DCNA,	Allisan	P
Ditranil	Botran	P
	Resisan	P
Dinocap	Crotothane	P
Dodemorph	Milban	P
	Karathane	P
Etridiazole (ethazol)	Truban	P
	Terrazole	P
Fenaminosulf	Lesan (Dexon)	P
Ferbam	Carbamate	P
	Ferbam	P
	Trifungol	P
	Others	P
Folpet	Folpan	P
	Phaltan	P
Iprodione	Chipco 26019	P
Mancozeb	Dithane M-45	P
	Mancozeb	P
	Manzate 200	P
Maneb	Maneb	P
	Dithane M-22	P
	Others	P
Metalaxyl	Subdue	S
Oxycarboxin	Plantvax	S
PCNB	Fungiclor	P
	PCNB	P
	Terraclor	P
Piperalin	Pipron	P
Polyoxin	Polyoxin	P
Polyram	Polyram	P
	Others	P
Sulfur	Many names	P
Thiophanate	Topsin M	S
	Others	S
Thiram	AAtack	P
	Arasan	P
	Tersan 75	P
	Thiram 75	P
	Others	P
Triadimefon	Bayleton	S

Table 20 continued

Common name	Trade names	Action
Triforine	Funginex	S,P,E
	Saprol	S,P,E
Vinclozolin	Ronilan	P
	Ornalin	P
Zineb	Dithane Z-78	P
	Zineb 75	P
	Others	P

* These are trade names of fungicides used in the nursery industry. Not all of these fungicides are labeled or recommended in all states. This list is not intended to be a complete list of all fungicides used in nurseries. There are numerous products which are combinations of two or more compounds. These are not included in this list.

** Systemic = S; Protectant = P; Eradicant = E.

Types of Fungicides

Chemical control of most fungal diseases depends on the application of fungicides before the pathogen arrives. Fungicides used in this way are said to be protectants (Table 20). Most broad spectrum fungicides are of this type. Protectants prevent fungal spores from germinating or kill them as they germinate, prior to penetrating the plant surface. These fungicides are ineffective after infection occurs except to reduce new infections. A few fungicides destroy pathogenic fungi after infection has occurred. These fungicides are called eradicants or therapu-tants. Their effectiveness is limited to a period of a few hours or days following infection. This post infection control is referred to as "kickback" action. An exception is the eradication of powdery mildew with sulfur.

Most protectant fungicides are not absorbed by the plant and are not translocated within the plant. They form a protective barrier over the surface of the plant to prevent infection. Some redistribution of these fungicides may occur during rainy periods but the effectiveness of the fungicide is dependent on thorough coverage during application. They must be applied and dry on the plant before they are exposed to rain or overhead irrigation and they must be present on the plant surface when the pathogen arrives. Captan, mancozeb, Daconil 2787® and thiram are examples of non-systemic protectant fungicides.

Recent technology has produced several fungicides that are absorbed and translocated within the plants. This type of fungicide is called a "systemic". Even though systemics are translocated to new growth, they are essentially protectant fungicides. Most systemics cannot kill fungi which have established themselves inside a plant. Benlate®, which is effective against a number of the Ascomycetes and Deuteromycetes, and Subdue® which is effective against members of the Phycomycetes, are examples of systemic fungicides. A number of the systemic fungicides also have kickback action. Bayleton®, for example, is effective against the apple powdery mildew pathogen for up to 48 hours after infection.

Protective fungicides are washed from the sprayed plant surfaces over a period of several days by rain and/or overhead irrigation and new unsprayed growth may emerge from buds. Protective fungicides, therefore, need to be applied several times at appropriate intervals during the period when the pathogen is active and/or the host is susceptible.

The nurseryman should understand the action of a fungicide he is applying to use it most effectively. Protectant fungicides are normally applied on 7 to 14 day intervals, but more frequent applications may be necessary in periods of heavy rains or frequent overhead irrigation. Systemic fungicides, on the other hand, are effective during wet or dry periods since they are translocated within the plant. Table 20 shows the action of some fungicides.

Acquired Resistance to Fungicides

The observance of resistant strains of fungal pathogens has occurred mainly with the use of narrow spectrum fungicides. Most of the systemic fungicides are of the narrow spectrum type and new fungicides in the future will probably be of this type. Their mode of action on the fungus is on a single metabolic pathway. Resistant fungal populations develop after repeated applications of a narrow spectrum fungicide due to a slight mutation in the pathogen. Resistance may have been present in a very small percentage of the fungal population and these resistant individuals can multiply in the presence of the fungicide and disease can develop. Using low rates of the fungicide increases the probability of resistant strains developing in fungal populations.

The best known examples of resistant fungi are strains of the apple scab fungus, *Venturia inaequalis*. Repeated applications of benomyl alone results in resistant strain development within 2 years in apple orchards. Other examples of fungi resistant to Benlate® are *Botrytis cinerea* (gray mold of a number of plants) and several species of powdery mildew fungi.

Benlate® is not the only fungicide for which resistance has been observed. *Phytophthora* sp. and *Pythium* sp. may become resistant to Subdue®. A partial list of fungicides for which resistance has been reported is in Table 21.

Resistance has not been reported against broad spectrum fungicides in use under field conditions. These should be used where repeated applications are necessary to control a disease. It is becoming a common practice to use a mixture of a broad and a narrow spectrum fungicide which provides the advantages of a systemic fungicide without the possible production of resistant strains of the pathogen. Resistant strains of *Venturia inaequalis* can be avoided in apple orchards with the use of Benlate® plus Captan mixtures. Similar results can be achieved by alternating sprays of Benlate® alone followed by Captan alone.

Table 21. Fungicides for which Resistance has been Reported

Common Name	Trade Name*	Resistant Pathogen**	Disease
Benomyl	Benlate	*Botrytis cinerea*	Gray mold
		Coccomyces hiemalis	Cherry leaf spot
		Erysiphe cichoracearum	Powdery mildew
		Fusicladium effusum	
		Monilinia fructicola	Peach brown rot
		Sclerotinia sp.	White mold
		Sphaerotheca fuliginea	Powdery mildew
		Uncinula necator	Powdery mildew
Carbendazim	Bavistin	*Penicillium expansum*	
Iprodione	Chipco 26019	*Botrytis cinerea*	Gray mold
	Rovarol	*Botrytis tulipae*	Tulip fire
		Monilinia fructicola	Peach brown rot
Metalaxyl	Subdue	*Pythium* sp.	Root rot
		Phytophthora sp.	Root rot
Vinclozolin	Ronilan	*Botrytis tulipae*	Tulip fire
	Ornalin	*Monilinia fructicola*	Peach brown rot

* This is not a complete list of all fungicides for which resistance has been reported. There may be several trade names for a particular fungicide. The above names are examples only and do not imply any criticism or endorsement of the particular products named.

** The listing of these pathogens does not imply that they are resistant in all growing situations.

The problem that resistant fungal populations represent cannot be over emphasized to the nurseryman. Many of the fungicides for which resistance has been reported are the best control chemicals for particular diseases. However, when a resistant population of a fungus is causing a disease, these fungicides will be of no value.

Fungicide Toxicity

Fungicides are designed to be toxic to fungi. Fungicides are usually toxic (phytotoxic) to plants as well. Even those which are labeled for a particular plant species may be toxic if used at rates higher than recommended on the label. Fungi are generally more sensitive to fungicides than are ornamentals. This differential toxicity of fungicides makes them valuable in the nursery.

Fungicides are generally less toxic to animals and less harmful to the environment than other pesticides. The mammalian toxicity of the active ingredient of a fungicide is clearly printed on the label of the product. The toxicity is reported as LD_{50} (Lethal Dose for 50% of test animals) in milligrams of active ingredient per kilogram of body weight of the test animal. The values will normally be reported for oral and dermal (skin) toxicities.

Toxicity ratings are normally determined for rats, mice, dogs or rabbits. The higher the LD_{50}, the less toxic the compound is to mammals that were tested. Of course, these toxicity ratings cannot be determined directly for man, but the relative toxicity can be determined from lower animals. For example, the acute oral LD_{50}'s for rats of Captan, PCNB and Subdue® are 10,000, 1,700 and 669 mg/kg body weight, respectively. Clearly, Subdue® is more toxic than the other two and PCNB is more toxic than Captan. In comparison with insecticides, none of these three fungicides is as toxic as the insecticide lindane which has an LD_{50} of 88-125.

Fungicide Formulations

Fungicides are produced in a variety of formulations and a single compound is frequently found in several forms. They are marketed as emulsifiable concentrates (EC), flowables (F), wettable powders (WP), dry flowable (DF) and dusts (D). Emulsifiable concentrates are liquids with the active ingredient dissolved in a solvent which will mix with water to form an emulsion. The flowable fungicides are thick water suspensions containing the active ingredient which is further diluted when the produce is added to water in the spray tank. Wettable powders are fine powders which contain the active ingredient of the fungicide and, when placed in water, they remain in suspension with constant agitation. Dry flowables are fine granules and when put in water remain suspended. Dusts are fine powders which are applied in the dry form.

Prior to the advent of modern formulation techniques, many fungicides were relatively crude preparations which were difficult to keep in suspension in spray tanks. Wetting agents were necessary to facilitate the mixing of the compound with water and to cause the spray to spread over and adhere to plant surfaces. Most fungicides produced today are mixed with additives (spreaders, stickers or wetting agents) by the manufacturer, making further additives unnecessary. Unless the product label specifically states that additives are necessary, the grower is best advised not to use them. The effectiveness of the fungicide may actually be reduced by an excess of additives or by the addition of the wrong type of additive.

Table 22. Fungicide Disease Applications on Nursery Plants

Fungicide* (Trade Names)	Disease Application: Anthracnose	Botrytis Blight	Damping-off & Root Rot (by Phycomycetes)	Damping-off & Rot (not by Phycomycetes)	Downy Mildew	Leaf & Twig Blight	Leaf Spot & Needlecast	Petal Blight	Powdery Mildew	Rust	Scab
Acti-dione								X	X		
Arasan			X	X							
Banrot			X	X							
Bayleton								X			
Benlate	X	X		X		X	X	X	X		X
Bordeaux Mix	X	X			X	X	X	X		X	X
Botran		X		X		X		X			
Captan	X	X		X		X	X	X			X
Copper	X	X			X	X	X	X			X
Chipco 26019		X									
Cyprex	X				X	X	X	X			X
Daconil 2787	X	X		X	X	X	X	X	X	X	X
Difolatan	X					X	X	X			X
Dikar									X	X	X
Dithane M-22	X	X	X			X	X	X			X
Dithane M-45	X						X	X			X
Dithane Z-78	X				X	X	X	X		X	X
Exotherm Termil		X									
Ferbam	X	X	X	X	X	X	X	X	X	X	X
Fongarid			X								
Fore	X	X					X	X		X	X
Funginex									X		
Karathane									X		
Lesan			X								
Maneb	X	X	X			X	X	X		X	X
Mancozeb	X						X			X	X
Manzate D						X	X	X		X	X
Manzate 200	X	X	X				X	X			X
Milban									X		
Mildex									X		
Ornalin		X									
Orthocide	X	X				X	X	X			X
PCNB				X				X			
Phaltan	X	X	X	X	X	X	X		X		X
Pipron									X		
Quintar								X			X
Subdue			X								
Sulfur	X					X	X	X	X		X
Terraclor				X				X			
Terrazole			X								
Thiram			X	X						X	X
Triforine									X		
Truban			X								
Zineb	X				X	X	X	X		X	X

* This list is for reference only and does not represent an endorsement nor imply criticism of the products.

Selecting a Fungicide

The nurseryman must obtain an accurate diagnosis of a fungal disease and its pathogen before selecting a fungicide. As stated previously, diseases caused by members of the different classes of fungi may require particular control measures with specific fungicides. Once the disease and its pathogen have been identified, an effective fungicide can be selected.

Hundreds of fungicides are available on the market today. A grower may be confused about which ones to stock and use. No single fungicide is labeled for all plant species and fungal diseases that are found in nurseries. It is difficult to prepare a fungicide guide for all diseases on all plants and all possible fungicides to cover every situation that will be encountered by the grower.

General information concerning fungicides and their disease applications can assist growers in selecting fungicides. A list of fungicides commonly found in nurseries and the disease types for which they have been used is in Table 22. The table can be used, for example, to determine that Lesan® controls damping-off and root rots caused by Phycomycetes but is not useful for the control of most other types of diseases. The table is for reference only and does not represent an endorsement or imply criticism of the products listed. Specific disease information can be obtained from the labels of individual fungicides.

Timing Fungicide Application

All nursery plants are susceptible to their own particular diseases. Generally, fungicides are used as a preventive measure to protect against those diseases which are common to that particular plant. They may also be applied to halt the spread of a disease from infected plants to healthy plants.

Timing the application of a fungicide is the most important factor determining the effectiveness of the control of a disease. Most fungi are vulnerable to fungicides in particular stages of their life cycles. These periods of vulnerability generally occur during the growing period of the nursery plants. To be effective, the fungicide must be applied within the period of vulnerability of the pathogen. This period often occurs when the host plant is in a susceptible growth stage and environmental conditions are favorable for spore production by the pathogen. Fungicide applications can begin just prior to and continue during this period to provide protection. If this period is not known for a particular disease, fungicide applications may be too early or too late to be effective.

Nurserymen must have some knowledge about the life cycle of the pathogen to control it with a fungicide. For example, *Venturia inaequalis*, the apple scab pathogen, is vulnerable to fungicides during and immediately following rainfall. During drier periods, fungicides are not necessary to control this pathogen because spores of the fungus are not germinating. (In arid parts of the world where apples are grown with the aid of irrigation, apple scab is not a problem.)

The most reliable information concerning plant pathogenic fungi and their times of vulnerability to fungicides is available from plant disease specialists. These specialists have determined through research and experience when applications of fungicides are most effective in the control of plant diseases.

The rate and frequency of fungicide application are influenced by the properties of the individual product. These factors have been determined by the manufacturer and are printed on the label of the product. Fungicides are most effective in the control of plant diseases when they are used at the rates specified on the label. Rates in excess of those recommended on the label may cause phytotoxicity, and rates less than those recommended may be ineffective in the control of a particular disease and may encourage the development of fungal resistance to the narrow spectrum fungicides.

The grower must exercise caution and apply fungicides only to those plants which are specified on the label. The use of a product on plants which are not on the label may result in damage to these plants.

Methods of Application

The method of application is another important factor determining the effectiveness of a fungicide. Most fungicides are in a form suitable for mixing with water. These are usually applied with a sprayer of some type. Several types of sprayers are available and are discussed below.

Small hand sprayers operated by compressed air are most commonly used in small nurseries. They are excellent for close work in sales yards or greenhouses.

Mechanical or power-driven sprayers are used for covering large areas. Most of these ground sprayers are operated on a tractor. The majority are high-pressure sprayers that use a pump which can produce sufficient water pressure to distribute the spray mixture. Many have an agitator to insure that the compound remains sufficiently dispersed in the water in the tank. The spray from these sprayers is dispersed in small droplets to provide thorough coverage.

Air-blast sprayers are becoming more popular in nurseries. These use a low-pressure pump to force the water through nozzles. A fan provides large volumes of high-speed air which distributes the spray in very fine droplets. These sprayers lack the directional capability of high-pressure nozzle boom sprayers.

Another type of sprayer is the mist blower. This type of equipment provides ultrafine droplets of spray that penetrate dense foliage canopies and provide uniform coverage of entire plants. These are particularly effective when spraying evergreen plants like *Arbovitae* spp. and *Juniperus* spp.. Mist blowers are available in ground rigs and backpack units.

Recent developments in sprayers and nozzles has lead to the development of low volume (LV) and ultra low volume (ULV) sprayers. These sprayers deliver a highly concentrated spray over the plant foliage. The primary advantage of such sprayers is that they require less water than conventional sprayers to cover a given area. This results in fewer trips to refill tanks.

While conventional sprayers apply the fungicide to "run-off", LV and ULV sprayers provide full coverage of the foliage without run-off and thus avoid loss of fungicide on the ground. A major disadvantage of the low volume sprays is that the droplets are so small that they remain suspended in the air for long periods of time. While this is desirable for penetration to occur, it permits the spray to drift to non-target species on slight currents of air.

Two additional methods of fungicide application are drenches and dips. Dips are used for many types of plants and plant products prior to storage. Drenches are used for plant beds and containers to control root diseases. This is the primary method of application for soil-borne diseases such as Phytophthora root rot. This method of application is very expensive by hand and may be applied through the irrigation system in the future.

A relatively new application method to the nursery industry is in-line treatment of plants with fungicides. Although not very widely used, the addition of fungicides to irrigation or misting water has many possibilities for nursery growers. Laws governing this type of application vary from state to state.

Systemic fungicides could easily be applied to field and container-grown plants. Fungicides designed to kill soil-borne pathogens could be applied in this manner. Additional uses of in-line treatment could be in cutting bed mist systems where foliar fungicides are desirable on rooted plants that must be held for a year or more. Fungicides must be very carefully chosen for use during propagation.

Several complications present themselves with in-line fungicide application. Rates are difficult to regulate and phytotoxicity may result where irregular watering patterns occur. The fungicides used in these systems must be soluble, flowable or emulsifiable concentrates. The emulsifiable concentrate compounds must be in non-corrosive carriers or solvents. Rapidly developing technologies will overcome these complications of in-line systems as irrigation systems become more common in the nursery industry.

Compatibility

The costs of labor and fuel required to apply fungicides to nursery fields is a constant concern to growers. One method to reduce these costs is to mix several pesticides in one tank (tank mix) and reduce the number of passes necessary over a field. Unless the grower is familiar with the tank mix he is using, this can result in more harm than good.

Not all fungicides are compatible with other fungicides, fertilizers or insecticides. Most will have a statement on the label indicating their compatibility with other insecticides. The grower may resort to a compatibility chart to determine if two pesticides may be mixed.

Several things can happen when non-compatible pesticides are placed together in a spray tank. The most noticeable is that the compounds combine chemically and precipitate out of suspension. When this happens, the precipitate collects in the bottom of the tank in a thick paste that cannot be distributed by the sprayer. Other effects of incompatibility include reduced efficacy resulting in poor disease control and phytotoxicity.

Many fungicides can be mixed with a number of other fungicides and insecticides. However, fungicides should never be mixed with herbicides. Unless the grower is familiar with a particular tank mixture, he should rely on information from pesticide specialists to decide on the use of these.

Fungicides in the Future

Research, development and formulation of fungicides are very costly processes. A conservative estimate of the costs of these procedures for a single compound is in excess of 15 million dollars. To recover these costs, manufacturers have concentrated on production of fungicides for diseases involving large acreages.

Historically, fungicides used in the nursery industry have been those developed for field crop application. Few fungicides are used exclusively on ornamental plants. Many manufacturers are expanding the labels of their products to include nursery plants. Growers can expect to see more fungicides for nursery plants as the ornamental industry becomes larger and more important to agriculture in the United States.

Unique fungicidal compounds and new formulation techniques will provide newer, more powerful fungicides to the nurseryman in the future. Innovative application techniques can also be expected.

Systemic fungicides which do not promote resistant strains are being developed. These will require fewer applications while still protecting new growth.

Several new techniques of application are being tested. One promising system recirculates the spray which is not deposited on the plants and uses it again. Recirculation results in less fungicide being lost on the ground. Another new application technique uses electrically charged spray droplets that adhere to the plant surface to insure thorough coverage.

Nurserymen can expect to gain better control of fungal diseases when the newer fungicides are marketed. However, they will never be able to rely exclusively on fungicides to protect or cure their plants. Disease-free liners, resistant varieties and meticulous sanitation procedures will always be necessary in combination with fungicides to provide a total disease management strategy.

Fumigants and Nematicides

R. C. Lambe, N. A. Lapp, C. Hadden and R. K. Jones

Chemicals applied to the soil to eradicate pathogenic fungi, bacteria, soil-parasitic insects, nematodes and weed seed are called fumigants (see Table 23). The economical use of fumigants is determined by previous crop loss and the value of the crop to be grown. The high value of certain ornamentals makes fumigation feasible.

Nematicides are fumigants or non-fumigant chemicals that are used specifically to control populations of plant parasitic nematodes. The economical use of nematicides is determined by analysis of plant roots and soil samples for plant parasitic nematodes. Nematicides recommended for ornamental plants are both fumigant and non-fumigant (contact) based on formulation, volatility and method of application. Fumigants are toxic to established plants, whereas the non-fumigant nematicides are non-toxic to some plants and can be applied safely around their roots.

Toxicity of volatile fumigants is dependent on their ability to move in the soil and reach propagules of plant pathogens. This movement is influenced by the chemical and absorptive characteristics of the fumigant, temperature, moisture, organic content, soil texture and variability in the soil profile. Container media consisting of large amounts of organic materials are difficult to fumigate and will require larger amounts of chemicals than non-organic ones. The activity or toxicity of fumigants is influenced by their chemical, physical and biological properties or a combination of the three.

Fumigants are usually applied in the liquid phase and volatilize to the gaseous phase. The rate at which the chemical leaves the liquid phase is dependent on its vapor pressure and this will determine movement in the soil. Chemicals with high vapor pressure, like methyl bromide (MB), move rapidly throughout the soil pore spaces by mass flow and by diffusion. Because diffusion is unaffected by gravity, physical barriers like plastic tarps are necessary at the soil surface to retain gases long enough to give maximum toxicity to plant pathogens.

Soil Temperature

Temperature of the soil is important because it affects the movement of gas. A rise in temperature will increase the vapor pressure of halogenated hydrocarbons like ethylene dibromide (EDB) and decrease their solubility in soil water. An increase in temperature also will increase the rate of other chemical reactions in the soil that influence chemical degradation and reduction in the effectiveness of its biological activity.

Fumigant Application

Fumigants are more volatile at higher temperatures and generally should be applied when the soil temperature is 55° to 85°F at the depth of injection (usually 6 inches). When fumigating cool wet soils, allow a longer waiting period before planting to avoid crop injury from fumigant residues. Highly volatile chemicals like MB must be sealed into the soil. This is usually accomplished by covering the soil with a plastic film before or immediately after the fumigant is applied. Special machines are available for injecting fumigants and laying plastic over large areas. With less volatile fumigants a sufficient surface seal can be attained by wetting, lightly packing or dragging the soil surface.

Soil Moisture

Moisture in the soil affects the activity of gaseous chemicals by its influence on their movement. They will pass readily through dry soil but are impeded in water-saturated ones. Highly volatile chemicals like MB are most effective against pathogens when they are moist but not saturated with water.

Fumigation is most effective in soils when the moisture level is slightly below field capacity but adequate for seed germination. When soil moisture reaches field capacity, water fills the air spaces between soil particles and prevents fumigant movement. Extremely dry soils, especially sandy types, allow fumigants to escape to the air before they have sufficiently penetrated the soil and killed the pests.

Physical Condition of the Soil

Physical factors like soil texture and composition influence fumigation effectiveness. Clay and higher organic soils will adsorb fumigants and, therefore, require higher rates for pathogen control. Moisture levels are more critical in clay soils than sandy ones, since the air spaces are smaller and more easily blocked by water. Cultivation of clay and highly organic soils may be necessary after fumigation to enhance dispersion of volative residues and prevent crop damage.

Soils should be well pulverized and free of clods when fumigants are applied. Fumigants will not penetrate the center of soil clods or thick layers of crop residue. Cover crops or other crop residues, therefore, should be plowed under well in advance of fumigation to allow decomposition.

Table 23. Treatments for Soil in Plant Beds and Potting Mixes

Material	Rate of Use	Application	Relative Effectiveness: Weeds	Nematodes	Insects	Bacteria	Fungi	Use, Remarks
steam	Heat soil to 180 to 200°F (30 min) 6 inches deep	Perforated pipes on or in soil, cover with tarp.	good	good	good	good	good	All crops, all pests.
aerated steam	145-160°F for 30 min	Same as steam	good	good	good	good	good	All crops, all pests.
dry heat	180°F for 30 min	place small quantities in oven.	good	good	good	good	good	
methyl bromide	2 lb/100 sq ft	Release in dishes spaced 30 ft apart under plastic cover. Fumigation period 1-2 days, aerate 1-2 weeks.	good	good	good	good	good	Use restricted to all plant production, and some crops.
	1 lb/100 sq ft		fair	good	good	poor	fair	
Vorlex	11 to 16 oz/100 sq ft (35-50 gal/acre)	Inject 4-6 inches deep, space chisels 8 inches apart, cover with plastic 7-15 days. Aerate 2-4 weeks.	good	good	good	fair	good	All crops, all pests. Long waiting period after fumigating in cold soil.
Vorlex (plus)	3-5 oz/100 sq ft (10-15 gal/acre)	As above, don't cover with plastic, seal fumigant with drag.	good	good	good	poor	fair	Ditto, uses of herbicides much restricted on label.
DD, Telone	8 to 15 oz/100 sq ft (25-50 gal/acre)	Inject 4-6 inches deep, space chisels 12 inches apart, cover 1 week. Aerate 3 weeks.	poor	good	fair	poor	poor	Not for greenhouse use.
chloropicrin (100%)	35-46 gal/acre	Inject 4-6 inches deep, space chisels 8 inches apart, cover with plastic 7-15 days. Aerate 2-4 weeks.	good	good	good	good	good	All crops, all pests. Long waiting periods in cold soils.
methyl bromide 67% plus chloropicrin	250-350 lb/acre	Inject 6-8 inches deep, space chisels 8 inches apart, cover with plastic tarp 1-3 days. Aerate 2 weeks.	good	good	good	good	good	Plant production beds strawberry and ornamentals.
Vapam	32 oz/100 sq ft	Aerate 2 weeks	fair	good	good	poor	fair	

Fumigants perform differently against various pathogens. Toxicity depends on the fumigant remaining in contact with the target organism for sufficient time and in sufficient concentration. There are large differences in the response of different pathogens to the concentration and time of fumigants. The stage of development of an organism also influences its resistance to a fumigant. Fungi like *Phytophthora* spp. and *Pythium* spp. are more sensitive to MB than *Fusarium* spp., *Sclerotium rolfsii* and *Verticillium albo-atrum*. Mixtures of MB and chloropocrin (CP) are more effective against *Verticillium albo-atrum* than MB alone. Fungus propagules imbedded in plant tissue are more resistant than those that are free in the soil. Fungi that form sclerotia are more resistant in general than those that do not.

Fumigants like MB are non-selective in their toxicity against fungi, and beneficial mycorrhizal fungi are much more sensitive than most soil-borne plant pathogens. This fact should be considered in evaluating plant growth following fumigation. It is unlikely that the concentration of a fumigant can be reduced to accommodate the mycorrhizal fungi and still eradicate pathogens.

Nematodes in the soil have been found to be more sensitive to fumigants than fungi or bacteria. More fumigants are useful for control of nematodes than for control of fungi and bacteria. Larvae of nematodes are, in general, more sensitive than adults.

Most fumigants must be applied prior to planting. Usually, a waiting period is required between the time the fumigant is applied and the time the crop is planted. This waiting period can be from a few days for MB to several weeks for Vapam and Mylone, depending on the type of fumigant, rate of application, soil type, soil moisture and temperature. Soil conditions for fumigation are usually better in the fall than spring, because the soil is warm and usually moist.

Fumigants

Telone, EDB, DD, Vapam and Vorlex. These chemicals are liquids which turn to gases very slowly once injected into the soil. They do not need to be applied under plastic, but the soil must be packed or sprinkled with water following injection to keep the chemical in the soil long enough to be effective. All of these have a moderate to low toxicity to man and present less hazard to the user as compared to MB and chloropicrin (CP).

Methyl Bromide. This chemical is packaged either separately or mixed with CP under pressure as a liquid. When released in the soil, it becomes a gas very quickly and must be injected under a plastic soil cover. Methyl bromide has a broad range of activity against insects, fungi, weed seeds and nematodes; however, its cost limits its use to high value crops like bedding plants grown from seed or azaleas or rhododendrons from cuttings. Methyl bromide is an odorless, tasteless gas and often has CP (teargas) added to the formulation (2%) for application protection. Chloropicrin also has nematicidal and fungicidal activities and is frequently combined with methyl bromide (MBCP) at a higher rate (33%). Methyl bromide, CP and combinations of these chemicals are nematicidal.

Mylone (Mico-fume) is available as a dust applied as a preplant fumigant. It can be spread over the soil surface with a fertilizer spreader and mixed into the upper 4 to 6 inches of soil by tilling.

Non-fumigant Nematicides

This group of chemicals, which does not have the broad spectrum activity of the fumigant nematicides, includes ethoprop (Mocap), fensulfathion (Dasanit), oxamyl (Vydate) and aldicarb (Temik). In contrast to the fumigants, these chemicals are not toxic to plants at the recommended usage rates and are often applied at planting time or after the plants are established. These chemicals are very toxic to man and animals and must be handled and applied very carefully with strict adherence to the label directions. Three of these chemicals (Mocap, Dasanit and Temik) are "Restricted Use" pesticides.

Mocap EC Insecticide and Nematicide (Restricted Use). This is a concentrated liquid formulation which is to be mixed with water and can be used as a bare root and tuber dip or a pot, bed, bench and field drench treatment. There are a number of ornamentals listed on the label for which it is safe to use Mocap. If plants other than those mentioned on the label require treatment, only a few plants should be treated until the effects of this chemical on the plants can be determined.

Dasanit 15 Percent Granular (Restricted Use). This granular nematicide-insecticide is registered for use as a preplant treatment of beds, benches and potting soil. Again, the label indicates a number of plants on which this chemical can be used safely and lists several plants that should not be treated with this chemical as plant injury may result.

Temik 10 Percent Granular Aldicarb Pesticide (Restricted Use). This chemical has received a special label for use on ornamentals in North Carolina and several other states. It can be applied to a wide variety of plants which are listed on the label and has good nematicidal activity. There are certain restrictions as to the waiting period before these plants can be sold. This material is very hazardous to the applicator but has the best nematicidal activity.

Vydate L Insecticide/Nematicide. This chemical is not a "Restricted Use" chemical, but it is sufficiently toxic to man and animals that care should be taken during the handling and application. This pesticide can be used as a root, corn or bulb dip, soil drench, foliar spray or as a preplant soil treatment. The ornamentals for which it is registered are listed on the label.

All chemicals can be harmful to plants and animals if misused. The user should follow all label instructions whenever he is applying pesticides.

In addition to chemicals, soil may be disinfested with heat (Table 23).

Additional Literature

1. *Farm Chemicals Handbook,* available from Meister Publishing Co., 37841 Euclid Ave., Willoughby, OH 44094.
2. *The Insecticide, Herbicide, Fungicide Quick Guide,* by B.G. Page and W.T. Thomson, available from Thomson Publications, P.O. Box 9335, Fresno, CA 93791.
3. *Agricultural Chemicals, Book IV, Fungicides,* by W.T. Thomson, available from Thomson Publications above.
4. *Tree, Turf and Ornamental Pesticide Guide,* by W.T. Thomson, available from Thomson Publications above.

Strategies for Control of Diseases Incited by Bacteria and Mycoplasmalike Organisms

George H. Lacy

Bacteria and mycoplasmalike organisms (MLO) causing plant diseases are relatively simple and primitive organisms compared to pathogenic fungi and nematodes. These simple organisms lack several membrane supported organelles (mitochondria, endoplasmic reticula and nuclei) and the protein synthesis system that more complex organisms such as plants have. Because of these differences in physiology, it is not surprising that these organisms do not respond to pesticides designed for fungi and nematodes. This article will discuss some strategies available for control of the diseases incited by bacteria and MLO.

These diseases can be controlled either by applying chemical compounds to kill or inhibit the pathogens or manipulating the biology of the host or its environment for control. The first category is chemical control and the second is biological control. Examples of control tactics for each category will be considered briefly. Some of the examples are drawn from experimental procedures and therefore, are not yet recommended.

I. Chemical Control

1. *Disinfesting agents.* Disinfestation of tools and work surfaces with alcohol or sodium hypochlorite (liquid bleach) is effective in preventing the spread of fire blight (caused by *Erwinia amylovora*) on apple or pear by pruning wounds. It is also useful for disinfesting potato seed piece cutters to prevent bacterial soft rot (caused by *Erwinia carotovora* subsp. *carotovora*) and preparing cuttings of ornamental plants to prevent spread of crown gall (caused by *Agrobacterium tumefaciens*).

2. *Acidic sprays.* Experimentally, the acid sensitivity of the organisms causing fire blight and pear blast (*Pseudomonas syringae* pv. *syringae*) has been exploited to control disease development. Acidic sprays of tartaric and citric acid were used.

3. *Fungicides.* Some fungicides have antibacterial activity. Those containing copper (Bordeaux mixture) or zinc (zineb) seem to be the most effective. Specifically, copper is effective against fire blight and zinc against bacterial spot of peach (caused by *Xanthomonas Campestris* pv. *pruni*). These metals are general biocides having several possible and probably coordinated modes of action against bacteria.

Captan has some activity for control of bacterial leaf spot of zinnia (caused by *Xanthomonas campestris* pv. *zinniae*). This fungicide is an halogen-containing compound.

4. *Fumigants.* For soil-borne pathogens such as those causing Granville wilt of tobacco (*Pseudomonas solanacearum*) or crown gall, fumigation of soil or planting materials with methyl bromide, or chloropicrin may be effective. These bromine or chlorine-containing compounds are general biocides. Although the cost of fumigation may be affordable in intensive ornamental culture, it is often too expensive for use in field culture.

5. *Antibiotics.* The most specific bactericides are the antibiotics in the streptomycin and tetracycline groups. Originally these compounds were derived from water soluble compounds produced by other bacteria (*Streptomyces* spp.). Both groups of compounds inhibit protein synthesis mechanisms typical of bacteria and MLO. The most effective use of streptomycin is to control fire blight of some fruit crops and bacterial spot of peach in orchards. Tetracycline, especially oxytetracycline, is also useful for control of fire blight and is used to control several MLO diseases, such as pear decline (causal agent not yet completely described).

A major drawback to the use of these compounds is that pathogen resistance arises very quickly. For example, on the West Coast, streptomycin is practically useless for control of fire blight since resistance is so widespread. On the East Coast, because of developmental differences in the disease, in cultural practices and orchard sizes, less antibiotic is used and streptomycin is still effective because the selection pressure for resistant strains is less intense. One may rather safely predict that streptomycin resistance will eventually appear on the East Coast by spontaneous mutation or physical spread of antibiotic resistant strains from the West Coast.

Additional antibiotic controls for bacterial pathogens must be developed and screened now so that if both streptomycin and tetracycline fail, new compounds will be available immediately to replace them.

6. *Insecticides.* Several diseases incited by bacteria and MLO are disseminated by insect vectors. Control of these diseases is often based on control of the vector rather than direct control of the pathogen. An example is Stewart's wilt of sweet corn (caused by *Erwinia stewartii*). Its control is based on using carbaryl to control the flea beetles that carry the pathogen from plant to plant.

II. Biological Control

Biological control is arbitrarily divided into three areas: host resistance, cultural practices and biological antagonism. Each area will be considered separately.

1. *Host resistance.* Host resistance depends on finding germplasm that is resistant to disease development. For bacterial diseases, no practical resistance to soft rot exists, some general resistance to fire blight exists, and good resistance to various bean blights and wildfire of tobacco exists. Effectiveness of host resistance seems to correlate with the specificity of the pathogen for its host(s). Soft rotting bacteria attack a very wide variety of hosts, the fire blight causal agent attacks several related hosts, but the bean and tobacco blighters attack only one host.

Disease resistance in a host is not always coexistent with the plant's best horticultural characters. In breeding programs, moving genes for resistance into genetic backgrounds compatible with host productivity is often made difficult by close linkage to non-desirable traits or ploidy problems. In the future, perhaps, recombinant DNA techniques will overcome these problems.

2. *Cultural practices.* Cultural practices offer the most complex assortment of control possibilities for diseases caused by bacteria and MLO. Disease can be partially controlled by using pathogen-free propagating materials, manipulating host nutrition, sanitation, crop rotation, irrigation and cultivation. Brief examples will be presented for each.

a) Pathogen-free propagating materials. Crown gall can be reduced significantly by using pathogen-free cuttings.

b) Host nutrition. Excess nitrogen fertilization often leads to more rapid or intense disease development. This is related to development of succulent and susceptible new growth and/or growth cracks developing in fruits, tubers or storage roots.

c) Sanitation. Sanitation for disease control may include several components: disinfestation of tools, equipment, personnel, as well as seeds and propagating material; roguing or removing and destroying diseased plants; pruning cankers that serve as inocula sources for secondary spread; and removal of weed host reservoirs for pathogenic inocula.

d) Crop rotation. Crop rotation with non-host plants may eliminate some pathogenic bacteria. It is variously effective for control of Granville wilt, black rot of cabbage, bacteria diseases of soybeans and others but not crown gall.

e) Irrigation. Excess moisture and overhead irrigation are primary means by which many pathogenic bacteria are spread from plant to plant.

f) Cultivation. Cultivation late in the growing season may result in flushes of succulent tissue susceptible to fire blight. On the other hand, bacterial overwintering can be reduced by cultivation-induced decomposition of plant debris during fallow periods. Cultivation may wound plants and provide points of infection.

g) Biological antagonism. Perhaps this is the most exciting area in plant disease control since several systems for biological antagonism of pathogens are currently being developed and some have been released for commercial use.

h) Bacterization. Bacterization is the process of inoculating seeds, seed pieces and roots with specific root-colonizing bacteria. Some of these bacteria cause enhanced growth and yields of the plants they inhabit. Some of these bacteria may also displace harmful microorganisms from the roots.

i) *Agrocin 84.* For crown gall, incited by *Agrobacterium tumefaciens*, a closely related, non-pathogenic bacterium *Agrobacterium radiobacter* has been discovered that in some instances produces very specific toxins against the pathogen. See the section on crown gall for more information.

Disease Resistance Among Woody Ornamentals

J. T. Walker

Resistance to diseases is the first line of a plant's defense. Were it not so, we might commonly experience complete plant devastation. Annually someplace in the southeastern United States, the proper environmental conditions, the pathogen and the susceptible host plant come together in the right combination for disease to develop. Sometimes it occurs quite often, yet some plants in commercial enterprises or backyard home gardens do survive the attack.

Genetically controlled resistance to plant diseases is mentioned throughout the agronomic, horticultural and pathological literature as one of the most important methods utilized for disease control. This is based on the successes resulting from crop breeding programs in plant science centers throughout the world—some of which are noted for their continuous contributions to developing new cultivars of corn, wheat, rye or other grains, or horticultural crops such as apples, peaches, beans, tomatoes or peas. A majority of these programs, as they should, have devoted their major efforts to developing greater yields, better quality fruit, more colorful flowers, greater hardiness, straighter or shorter stems and to some extent, attention to disease or insect resistance. This does not imply that the proper attention has not been given to developing agronomic cultivars or varieties with disease resistance, but it seems to have been a secondary consideration until it became a necessity because of economic pressures or disease epidemics.

Some of the breeding programs in ornamental horticulture probably grew out of the closely aligned programs in the fruit and vegetable industry that began in the mid-19th century and followed closely the development of the science of genetics. According to C. G. Patten, a pear and apple breeding program was in existence at Charles City, Iowa from the period of 1868 to 1932, and the one at the New York Agricultural Experiment Station (Geneva) began in 1892 and still remains active today.

Many of these projects closely paralleled the developing science of plant pathology, or were initiated because of the impact certain diseases were having on plants as in the case of fire blight of apple and pear, the first plant disease discovered to be caused by a bacterium. It is still one of the most destructive diseases of certain Roseaceous plants. Many woody ornamental species grown in the United States, Canada, Great Britain, Europe and New Zealand are susceptible, including 14 species of cotoneaster, flowering quince, hawthorn, Kerria, mountain ash and certain spirea. Research efforts led to developing lists of apple and pear cultivars susceptible, moderately resistant or resistant to fire blight (Table 24).

Table 24. Some Roseaceous woody ornamental plants which were resistant to *Erwinia amylovora* (bacterium causing fire blight) after artificial inoculation.

Cotoneaster bacillaris	*Prunus americana*
C. disticha	*P. cerasus*
C. harroviana	*P. hortulana*
C. newryensis	*P. persica*
Pyracantha coccinea	*Rosa californica*
cv. Shawnee	*Rosa gymnocarpa*
cv. Mojave	*Spirea prunifolia*
Photinia serrulata	

Meaningful information regarding species or cultivars of woody ornamental plants which are resistant to other diseases is not easy to locate. A search of nursery catalogs, experiment station bulletins, extension circulars and plant society literature provides scattered information. For example, The American Rose Society, in its annual listing and evaluation of hybrids, provides a disease/pest rating, but does not provide detailed facts on black spot, powdery mildew or rust, the most common diseases. This general listing takes these problems into consideration, but when specific answers to one pest are sought, then it is a matter of interpretation.

Crab apples are important components of urban landscapes, particularly where excessive tree height is not desired. However, one of the disadvantages to certain cultivars of crab apple is their extreme susceptibility to apple scab, cedar apple rust, and as mentioned earlier, fire blight. Unfortunately, these are the cultivars most nurseries produce. Les Nichols of Pennsylvania State University has worked diligently on examining and collating data on the susceptibility of crab apple cultivars to these diseases. Largely through his efforts, cities and towns can now utilize this information in their planting plans, and hopefully avoid costly spray programs which are necessary to retain leaves on susceptible varieties. Those cultivars resistant to scab are listed in the section on crab apple diseases in this bulletin.

Another serious disease of azalea and rhododendron is root rot caused by *Phytophthora* sp. or water molds. Fortunately, we have good resistance to this disease although most hybrids and species are susceptible. The following five rhododendron hybrids are reported resistant to *Phytophthora cinnamomi*: 'Caroline', 'Martha Isaacson', 'Pink Trumpet', 'Professor Hugo de Vries', and 'Red Head'. Others were of moderate resistance, and some rhododendron species such as *Rhododendron davidsonianum* 'Serenade', *R. delavayi, R. occidentale, R. pseudochrysanthum, R. poukhanense* and *R. sanctum* were more resistant. It was stressed that there was variability in the level of resistance among this latter group and more thorough testing should be performed. Seventy-three evergreen azalea cultivars in ten hybrid groups were evaluated for resistance to root rot caused by *Phytophthora cinnamomi.* See the azalea section for more information.

Powdery mildew is a general term for a common foliage disease of many woody ornamentals. It is more unsightly than it is harmful, and except in rare landscape situations, is seldom controlled by fungicide applications. During the late 1960s, Hibben et al. made repeated observations on lilac mildew found on various cultivars in northeastern locations of the United States. These results, published in 1977 provide landscapers and lilac breeders with information regarding the reation of many cultivars to the most common diseases of this popular horticultural plant. *Syringa vulgaris* cultivars were more susceptible than cultivars of other species, but there was considerable difference in susceptibility among the former group. The mechanism of resistance was not investigated, but practical information was provided from which plant breeders or hybridizers might glean an appropriate base for incorporating resistance.

Similar information might be drawn together on other plant species if only there was sufficient interest on behalf of the landscape or nursery industry to provide support for the compilation of such data. Very few breeding programs for woody ornamentals are in existence today. Baker and Linderman point out that the return is too small on breeding for disease resistance because ornamentals are a high value per acre crop, and growers are willing to spend more for chemical pest control. More realistically, resistant cultivars replace those that disappear from the trade because of their exteme susceptibility to disease. The susceptibility of *Photinia serrulata* to powdery mildew is part of the reason why it has been replaced in the nursery industry by *P. fraseri* and *P. glabra.*

Camellias are popular woody ornamentals in the Southeast and are subject to at least four main diseases. In evaluations at Clemson University, few cultivars of *Camellia japonica* were resistant to dieback and canker caused by *Glomerella cingulata.* Cultivars known to be resistant are: 'Woodville Red', 'Professor Sargent', 'Governor Mouton' and 'Cho-Cho-San'. Most *C. sasanqua* cultivars are not as tolerant of dieback as the *C. japonica* cultivars. Those that have some resistance include 'Mine-no-Yuki', 'Daydream', 'Maiden's Blush', 'Apple Blossom' and 'Setsugekka'. *C. sasanqua* cultivars are more resistant to root rot caused by *Phytophthora cinnamomi* than are cultivars of *C. japonica.*

Hollies comprise a considerable portion of our southern landscapes and are extensively grown in southern nurseries. Most are subject to a variety of fungus-caused leaf spots, diebacks, root rots and sooty-molds. As a group, the Japanese hollies (various cvs. of *Ilex crenata*) probably have fewer total diseases than the American (*Ilex opaca*) holly; the Yaupon holly (*I. vomitoria*) may have the least number. This may be the result of more research on *I. opaca* than on the other species. Several diseases, however, cause greater plant damage to *I. crenata* than other holly species.

In the last two decades increasing attention has been devoted to the effects of air pollution on plants, including woody ornamentals. Many of the investigations have dealt with fumigation of plants with specific atmospheric pollutants at dosages somewhat above ambient levels. The effects of single pollutants such as ozone, NO, SO_2, ethylene and fluorides at various dosages on growth and reproduction have been studied at a variety of locations. Investigations are beginning on the effects of combined components, as well as particulates and acid precipitation on plants. As a result there are no lists of plants which are susceptible to the common air pollutants.

An important consideration in selecting plant material in accordance with their reaction to pollutants is that there is considerable variation among cultivars as there is with other diseases. For example, azaleas vary in susceptibility to ozone. Also, the influence of environmental factors and general plant health should be considered as these effect plant reactions to pollutants.

Resistance to specific plant parasitic nematodes among plants is known to occur and is attributed to root-exudates, failure of nematodes to penetrate roots, or the presence of phenolics or similar toxic compounds naturally present in the plant. In certain instances, nematodes may penetrate the roots, but fail to develop to the egg-laying stage. Again, more information is available concerning the susceptibility or resistance of agronomic crops to nematodes than there is about woody ornamentals.

Recent studies have demonstrated the reproductivity levels of several nematode species on holly species and aucuba in microplot experiments in North Carolina. Only root knot and stunt nematodes caused serious stunting of aucuba and Rotunda holly. Burfordi, Rotunda and Yaupon hollies were resistant to a particular root knot species.

Although all 13 rose root-stocks tested were considered hosts for northern root knot nematode (*Meloidogyne hapla*) and lesion nematode (*Pratylenchus penetrans*), there are differences in host efficiency, especially with two root-stocks, Rosa canina 'Success' and 'Heisohn's Rekord', which proved to be poor hosts for the northern root knot nematode (*M. hapla*). *Rosa rubiginosa* reportedly had some resistance to both nematodes. Nigh tested 18 ornamentals grown in Arizona for susceptibility to two species of root knot (*M. incognita* and *M. javanica*) and lesion nematode (*P. penetrans*). Oleander was not infected by *M. javanica* or *P. penetrans* but was by *M. incognita.* Lantana was not infected by *P. penetrans.*

There are limitations to disease resistance. It is not the utopia of disease control. Not all plants can be custom-created to contain the genes responsible for conferring resistance to specific diseases. Then, it often is difficult to incorporate resistance to several diseases in one variety.

In spite of these limitations, there is a need for greater use of disease resistant plants, especially woody ornamentals, to avoid disease losses. As new management schemes are developed for future pest control, through integrated pest management or similar programs, knowledge of disease resistant plant species or cultivars will become imperative to providing healthy plants in a healthy environment. The wide diversity of woody ornamentals should offer an excellent opportunity to demonstrate that resistance can be an effective means of combating disease. The challenge is great, the rewards unlimited.

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Plant Disease Clinics

R. K. Jones and E. H. Moody

As soon as a disease appears in the nursery, correct diagnosis is the first step in the initiation of a disease control program. A major objective of this bulletin is to help nurserymen diagnose disease problems. Plant Disease Clinics are equipped to aid growers in disease diagnosis and may also include insect identification. Most agricultural universities operate a clinic staffed by one or more Extension Plant Pathologists. Your state clinic will provide a correct diagnosis to reduce disease losses, increase plant quality and ultimately increase profits. Your local agricultural Extension agent can provide specific information on the clinic in your state.

The most important step in obtaining a correct diagnosis from a Plant Disease Clinic is submitting an adequate sample representative of the disease. On the form provided by your state clinic, include such information as type of planting, common and complete scientific name, cultivar, date planted, chemicals (pesticides, growth regulators, fertilizers) and rates used. Whenever possible, submit several fresh, entire plants showing typical symptoms including roots and growing media or soil. Healthy plants are helpful for comparison. *Dead or dry plants cannot be diagnosed.* As soon as the plants or plant material is collected, it should be placed in a plastic bag and tied around the main stem of the plant just above the soil to keep the soil off the foliage. Submit the sample to the clinic as rapidly as possible. Because of numerous root diseases, it is necessary to submit several entire plants, including roots and soil, with woody ornamental plants. *The quality of the diagnosis will be no better than the quality of the sample.* For most fungal diseases, diagnosis may take only several days. Where complicated laboratory or greenhouse tests are necessary, as for bacterial, viral, mycoplasma and nematode diseases, it may take 3 to 6 weeks, depending on the organism involved and diagnostic analysis necessary to confirm the cause of the disease. The diagnosis and possible control measures will be sent back to you as soon as possible.

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Mycorrhizal Fungi as Deterrents to Disease

Donald E. Carling

Mycorrhizal fungi colonize (grow on and within) the roots of most plants, including most ornamentals, with the resulting association benefiting both the plant and the fungus. Many mushrooms and puff ball fungi are mycorrhizal. Although mycorrhizal fungi invade plant roots in much the same manner as do pathogenic fungi, they do not disrupt or kill root cells. Instead they harmlessly occupy space within or between root cells and assist the plant in its uptake of water and nutrients from the soil. Most studies involving mycorrhizal fungi have dealt with the improved absorption of nutrients and water, phenomena that are often observed in mycorrhizal plants. As a result the general effects of mycorrhizal fungi on plant growth and development are well documented (Mosse 1973).

More recently, the capabilities of mycorrhizal fungi as deterrents to plant disease have been studied. Many of these works are summarized in two excellent reviews (Marx 1972, Schench and Kellam 1978). It is not clear at this time if any general conclusions can be drawn from the combined work in this area. In several cases, however, mycorrhizal plants have been shown to be damaged to a lesser extent by root pathogens than are non-mycorrhizal plants.

It has been postulated that ectomycorrhizae (outside the plant root), the type found on pine, oak, willow, poplar and other trees, may protect roots by one of several mechanisms including: 1) utilizing root carbohydrates and other compounds that may otherwise be attractive to root pathogens; 2) providing a physical barrier to root pathogens in the form of the fungal mantle; 3) secreting antibiotics which inhibit or kill root pathogens; 4) supporting a protective rhizosphere population of other non-pathogenic microorganisms; and 5) stimulating the plant to produce chemical substances inhibitory to the development of root pathogens. Any one of these mechanisms, or several in combination, may account for the reductions in feeder root necrosis and increases in seedling survival that have been correlated with the presence of ectomycorrhizal fungi in the root systems of seedling pines.

Endomycorrhizae (inside the plant root cells), the type of mycorrhizae forming on the roots of nearly all plants other than the ectomycorrhizal plant groups mentioned above, also have a capability to reduce disease damage or disease development. With the exception of providing a physical barrier against pathogen attack, endomycorrhizae may utilize the same mechanisms as do ectomycorrhizae. Examples of endomycorrhizal mediated disease reduction include less plant stunting and reduced root infection in mycorrhizal strawberry plants infected by the fungus *Cylindrocarpon destructans*, and a reduction in disease damage from Fusarium wilt (*Fusarium oxysporum* f. sp. *lycopersicae*) infected mycorrhizal tomato plants.

A limited number of studies of this type have been conducted on ornamental plants. Shoot growth of poinsettia plants inoculated with an endomycorrhizal fungus prior to inoculation with *Pythium ultimum* and *Rhizoctonia solani* was equivalent to shoot growth in plants where no pathogens had been added. In this case, the positive actions of the mycorrhizal fungus neutralized the negative impact of the two pathogens. Mycorrhizal Japanese holly plants appear to be able to isolate more rapidly, or "wall off", areas in the root system infected by the black root rot fungus *Thielaviopsis basicola*, than non-mycorrhizal plants. This may explain the trend toward disease reduction observed in mycorrhizal Japanese holly plants. The application of commercially available *Pisolithus tinctorius*, an ectomycorrhizal fungus, is now a standard nursery practice in the production of seedling pines in fumigated beds to control Phytophthora root rot.

Although investigations to date suggest the general value of mycorrhizal fungi as agents of disease control, it is clear that much more study is required before general application of this knowledge will be possible. This is especially true in the case of woody ornamental nursery crops where most of the plants are produced from cuttings grown in "sterile" media. Much of the work to date in the disease control area has been on other types of crops. Further testing of mycorrhizal fungi on ornamental plants may in time permit us to identify mycorrhizal fungal species capable of performing effectively as biological deterrents to disease.

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Tissue Culture of Woody Ornamentals

John McRitchie

Tissue culture of flower and foliage ornamentals is presently enjoying great popularity largely because of the rapid clonal multiplication of highly desirable new varieties. Several nurseries maintain laboratories of their own while others purchase tissue cultured plants from independent laboratories or contract for the culturing of their own selected varieties.

More recently, with projected advantages of reduced cost, decreased production time and increased yield of forest crops, interest in tissue culture of woody perennial crops has increased.

Most woody ornamental plants are propagated vegetatively. Because of their slow growth habit, it is difficult to develop superior, uniform varieties from breeding programs. If such varieties were developed, considerable time would be required to propagate a significant supply of stock material. Thus, most improvement in varieties of woody ornamentals has resulted from the selection of chance mutations or sports.

This practice has drawbacks. As with all vegetative propagation, the danger exists for the propagation of plant pathogens along with the plant propagules. Tissue culture can be an effective tool in establishing pathogen-free propagation stock in the woody ornamental industry. This technique could be used to rapidly propagate the results of a breeding program or selection program as well as in propagating healthy plants.

Without question, the term "disease-free" has been misused. Only if plants are subjected to specific detection techniques for pathogens can they truly be called pathogen-free; and even then they may be free only from those pathogens for which they were tested. Tissue culturing does not guarantee "disease-free" results.

It is probable that viruses are carried along in the present vegetative propagation processes of numerous woody ornamentals. These viruses may produce no observable symptoms. Conversely, the desirable characteristics of certain selected clones may be the result of virus infection. Flower variegation in camellias is known to be caused by virus infection.

Virus diseases of roses have been a serious problem in the rose industry for many years. Recently an investigation of this problem in the rose industry in California uncovered several new virus diseases of rose. These viruses are transmitted to nursery plants by grafting infected rootstock and/or scions. Tissue culture techniques are being utilized to clean up propagation stocks and rapidly increase the supply of virus-free stock.

While the production of pathogen-free woody ornamental plants should result in advantages such as improved vigor and flower quality, it should be stressed that the resulting plants are still susceptible to plant diseases and should be protected accordingly. Tissue culture may be utilized more widely in woody ornamentals in the future.

Regulatory Control

Neil A. Lapp and D. J. Schweitzer

In order to prevent the movement of plant pests on nursery stock, laws have been established to require that nursery stock meet certain standards of freedom from diseases as well as from insects and other pests. These laws will vary from state to state and from country to country. Different requirements exist and must be met before ornamentals can be moved across national and international borders.

The laws do not prevent movement of pests which are not detected through a visual inspection. They are designed, through a series of inspections, to detect and prevent the movement of sizable infestations of common pests, unusual pests or pests specifically restricted by a given state.

Nursery Inspection

Most states require a minimum of one annual inspection during the growing season with more inspections as necessitated by turnover of nursery stock and the demands of the recipient state. Inspectors are trained to determine visually the presence of a pest on different types of plants. With proper training an inspector is also capable of determining the presence of suspected root problems (nematodes or fungi).

Inspections are generally based on a state's own standards of plant health; however, it is often necessary to modify these standards to meet the requirements of the state or country for which the plants are destined. Nurseries whose plants meet the inspection requirements will receive a plant inspection certificate which may take several forms. Many states issue a nursery license which can be duplicated by the nurseryman to accompany plant shipments, whereas others will issue inspection certificates to be used with each shipment. When out-of-state shipments are anticipated, the nursery inspector should be contacted to determine if specialized inspection is needed for shipment into those states.

Quarantines

Certain areas of the United States are subject to quarantines due to the presence of serious plant pests. These quarantines often put additional restrictions on nursery stock as they frequently restrict the movement of soil. The quarantines generally provide mechanisms whereby the nursery stock can be treated in a specified manner so it can be shipped out of the quarantined area. Some quarantines such as the Japanese Beetle Quarantine and the Witchweed Quarantine have been adopted by infested states and the U. S. Department of Agriculture and place stringent restrictions on the growing and movement of plant material from these areas. These *interior* quarantines provide mechanisms whereby nursery stock can be grown and shipped from quarantine areas. Nurserymen with questions regarding quarantines in their area that would affect them should contact their nursery inspector.

Another type of quarantine affecting the nurseryman is the *exterior* quarantine. This quarantine requires that plant material leaving infested states meet certain criteria established by non-infested states. These restrictions may vary considerably from state to state and region to region. When shipping nursery stock to markets in new states, the nurseryman should contact his nursery inspector to determine whether special restrictions are applicable to these areas.

Importing Plant Material from Foreign Countries

The U. S. Department of Agriculture has restrictions on the importation of plant material from other countries due to the occurrence of plant pests not known to occur in the United States. Certain plant material is completely prohibited (notably edible fruit and nut tree species) whereas other types are subject to lesser restrictions. Prior to importing plant material from other countries, the nurseryman should contact the nursery inspector to determine what restrictions are present and directions on obtaining proper permits for import.

Importing Plant Materials from Other States

Most states require that plant material being imported into their state be inspected in the state of origin and a certificate issued that attests to the *apparent freedom* of the plant material from injurious pests and diseases. However, this certification does not prevent the receiving state from taking action against a shipment of plants should they find evidence of actual infestation upon arrival of the plants in their state.

If the plant material you receive arrives with visible pest problems, a local nursery inspector should be contacted immediately so that remedial action can be taken. Some states have plant inspection stations along the state borders to inspect all plant material being imported. Most states rely upon spot inspections and the aid of importing nurserymen to prevent pest introductions. Most nurserymen desire to receive and sell pest free plant material.

Glossary of Terms

W. H. Wills

abscise—refers to the dropping of leaves or fruit through a rupturing of a special layer of cells at the base of petiole or fruit stalk.

acervulus, acervuli—a fungus fruiting body in which asexual spore-bearing structures occur in a layer exposed by the rupturing of host tissue at the surface.

antagonism—the phenomenon of one microorganism producing substances which inhibit the growth of some other microorganism/s.

anthracnose—a disease produced by one of several specific genera of fungi which results in characteristic, often zonate and distinctly marginate, spots on leaves, petioles, stems or fruits of the host.

antibiotic—a substance produced by a microorganism which in very low concentration inhibits or prevents growth of another microorganism.

apothecium, apothecia—a fungus fruiting body, sexually produced by a certain group of Ascomycetes, being open or saucer-shaped, on which sac-shaped structures bearing ascospores are borne.

ascomycete, ascocarp, ascospore, ascus—a group of fungi called *Ascomycetes* that reproduce by the sexual process, fruiting bodies called *ascocarps,* which bear on or within them, sac-like structures called *asci* (s. ascus) that contain the spores called *ascospores.*

biotic—of a biological nature; refers to a biological as opposed to an inanimate cause of disease.

blast—the killing of flower buds or fruit of plants.

blight—a general and rapid killing of plant tissue—sometimes used to describe specific diseases which produce a general destruction of tissue.

canker—a localized area of diseased plant tissue, sunken or raised, which usually involves destruction of the phloem, characteristically in stems of woody plants.

chlamydospore—a thick-walled fungus spore, usually microscopic, asexually produced, which functions as a resistant or overwintering stage.

chlorosis—yellowing of normally green plant tissue due to loss of chlorophyll.

cleistothecium, cleistothecia—a closed ascocarp which ruptures to release its spores—see Ascomycete. Produced by some powdery mildew fungi.

colonization—the period following infection during which a pathogen becomes established in its host (see infection).

conidium, pl. conidia—an asexually produced spore of a fungus, variously produced.

damping-off—the rotting off of the stem of a seedling plant (postemergence) or the rotting of the seed or seedling before breaking ground (pre-emergence).

decline—a disease condition of perennial woody plants in which slow death of the plant occurs over many months or years.

epidemic—widespread occurrence of a disease within a defined population [strictly speaking, a population of people (f. demos)]—the correct term to apply to a plant population is *epiphytotic.*

epiphytotic—see *epidemic* above.

ethylene—a gas commonly produced by some living plants, especially apple fruits, which has growth regulating properties in plants.

etiology—cause or causes of a disease.

fumigant—a chemical which, when applied to soil or other material to kill microorganisms present, volatilizes, releasing toxic gases which diffuse through the medium.

fungicide—strictly speaking a chemical which kills fungi; used to classify those chemicals which kill or inhibit growth and activity of fungi.

incidence (disease)—number of plants affected within a population—disease incidence should be distinguished from disease severity.

incubate—to allow a microorganism to grow undisturbed under a given set of conditions; also (incubation) the period of development of a pathogen within the host, or a period of rest of a pathogen prior to infection; the term has various uses and can be confusing.

infect (ion)—the process of establishing a physiological relationship between pathogen and host. Once infection has been effected, colonization begins—see *colonization.*

inoculate—to deposit a pathogen at the site of infection of a host (the infection court). Loosely and incorrectly used to describe the transfer of living cells of a microorganism to any place where they will grow and develop.

latent infection—the condition in which a pathogen is found in a quiescent condition producing no symptoms in the host.

lesion—a defined area of diseased tissue.

mildew—used several ways; to describe 1) the visible cobwebby body of a fungus; 2) disease caused by specific groups of fungi (such as powdery mildews, downy mildews); 3) the fungi which cause these diseases.

mold—similar to mildew; used to describe the visible evidence of certain fungi as well as certain diseases and the fungi which cause them (gray mold, white mold).

mosaic—a pattern of alternating discrete areas of green and yellow plant tissue, which may blend together; usually caused by viruses in the plant.

mycelium—the filamentous strands of fungus tissue which collectively make up the thallus or vegetative body of the fungus—see thallus.

mycology—the study of fungi.

mycoplasma—a microorganism, similar to a bacterium but lacking a cell wall; they may produce fungus-like filaments, hence mycoplasma.

mycorrhiza—literally fungus-root; an association of fungus and plant root in which the fungus grows on or in the plant root and may aid in the uptake of nutrients by the plant host.

necrosis—death, especially used as limited tissue death.

obligate parasite—any organism that requires its nutrients to be obtained from a living host.

oospore—among certain fungi, the oomycetes, the oospore is the result of the sexual process; it is analogous to the ascospore and basidiospore.

pathogen—any agent, biotic or abiotic, which causes diseases; commonly used by plant pathologists as a designation for the biotic agents.

penetration—pathogen movement or growth into a host plant or plant part.

perithecium—the sexually produced fruiting body of a group of ascomycetes characterized by presence of the asci in a body which releases the spores through a definite pore in that body.

phloem—the plant tissue between the woody core and outer layers consisting of cells which conduct the food produced in the leaves to other parts of the plant. Phloem is often the site of virus infection and is the tissue often affected in canker diseases.

predispose—usually used to mean to render a plant more susceptible to disease through unfavorable environmental conditions.

propagule—any discrete unit or body of a microorganism which is capable of growing into a new individual when separated from the parent body.

pycnidium, pl. pycnidia—an asexually produced reproductive body of certain fungi which bears conidia within its closed structure; may release the conidia variously.

replicate—the process by which a virus particle induces the host cell to reproduce the virus.

ring spot—a plant disease symptom characterized by small ring-shaped necrotic areas of leaf tissue with green centers; usually caused by a virus; the rings may be irregular or indistinct due to the pattern of small veins in the leaf.

rosette—a growth pattern of plants in which the leaves tend to be in a cluster very close together due to very short internodes; may be caused by some pathogens in some instances.

rust—any fungus of a group of basidiomycetes which are obligate parasites and produce rusty red or brown masses of spores on the host; also the disease caused by any one of these fungi.

saprophyte—a microorganism which gains its nutrients from dead organic matter.

sclerotium, pl. sclerotia—a compact mass of fungus tissue, often with a hard resistant rind surrounding a softer core.

severity—as in *disease severity,* the measure of damage done by a plant disease as distinguished from *disease incidence,* a measure of the number of individuals affected.

spiroplasma—a spiral-shaped mycoplasma-like (see mycoplasma) microorganism.

sporangium—a spore case, the body in which asexually produced spores of certain Phycomycetes are borne.

spore—a general term for reproductive propagules of many fungi; may be sexually or asexually produced and of a wide variety of size, shape and origin.

sporodochium, pl. sporodochia—a mass of fungus tissue rupturing through the host epidermis to the plant surface and bearing conidia and conidiophores in a cushion-like mass.

sporophore—literally a spore-bearer, therefore any of a number of simple or complex structures on which spores are borne.

stomate—the specialized pores in a leaf surface, most commonly on the lower surfaces; sometimes serve as points of entry for pathogens.

stroma—a mass of fungus tissue produced on its substrate, dead or living, and which bears some type of reproductive structure; often hard in texture.

symptom—the visible effect produced in a plant (or animal) by the presence of a pathogen.

suspect—a plant which is potentially subject to infection and colonization by a given pathogen.

tar spot—a disease of plants characterized by the presence of microscopic stromatic tissue at the surface of the host giving the appearance of a spot of tar.

thallus—the vegetative body of a fungus or of some of the lower plants.

variegation—alternating patterns of green, yellow or white plant tissue, usually genetic in origin.

vector—a living organism which serves as an agent for the transmission of a pathogenic microorganism.

virulent (-ence)—terms used with much imprecision; often used to indicate capacity to produce severe disease or to compare shades of difference in pathogenicity.

virus—specific particle of nucleic acid with a protein coat which, when introduced into a host, may cause disease symptoms.

wilt—a type of disease in which the plant host loses turgor and tissue collapses; verb indicating the action of wilting.

witches' broom—excessive proliferation of twigs or small branches in bunches resembling the straws of brooms; may be caused by mycoplasmas in woody plants.

zoospore—a motile (animal-like) asexually produced spore of certain fungi, especially Pythium and Phytophthora.

zygospore—the sexually produced spore of a Zygomycete produced by the fusing of nuclei of an antheridium and an oogomium.

COLOR PLATE I

1. Healthy boxwood roots (top) and Phytophthora rotted roots (bottom).

2. Boxwood branch dieback caused by Phytophthora root rot.

3. Internal boxwood stem discoloration caused by Phytophthora root rot.

4. Chlorotic azalea foliage caused by Phytophthora root rot.

5. Healthy azalea (left) and Phytophthora root rot (right).

6. Healthy azalea stem (left) and stem from Phytophthora affected plant (right).

7. Phytophthora root rot on rhododendron.

8. Healthy rhododendron roots (left) and Phytophthora rotted roots (right).

9. Kalmia leafspot.

COLOR PLATE II

1. Camellia flower blight (top) and cold injury (bottom).

2. Camellia flower blight apothecium.

3. Mosaic on camellia foliage.

4. Camellia flower color break.

5. Camellia leaf gall.

6. Photinia—Entomosporium leaf spot.

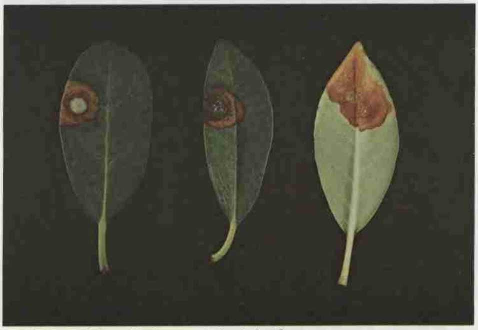
7. Chamaedorea palm—Phytophthora bud rot.

8. Liriope—anthracnose.

COLOR PLATE III

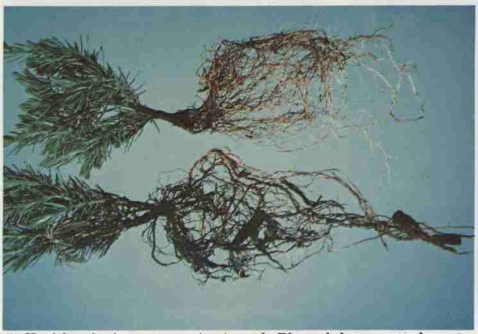
1. Rhododendron—Phytophthora dieback leaf symptoms.

2. Rhododendron—Phytophthora dieback stem discoloration.



3. *Colletotrichum* leaf spot of rhododendron.

4. *Rhizoctonia* stem blight of juniper liners.



5. Healthy juniper roots (top) and Phytophthora rotted roots (bottom).

6. Phytophthora root rot on juniper.

COLOR PLATE IV

1. Black root rot on Japanese holly (left) and healthy Japanese holly (right).

2. Black root rot on Japanese holly.

3. Black feeder root tips of Japanese holly.

4. Root-knot nematode galls.

5. Yaupon holly—Cylindrocladium leaf spot.

6. Camellia—algal leaf spot.

7. Rose-rust.

COLOR PLATE V

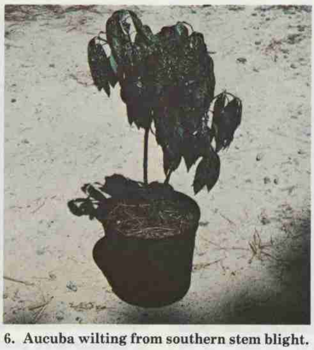
1. Azalea petal blight.

2. Blighted flower and sclerotia of azalea petal blight.

3. Botrytis blight or gray mold on azalea liner flowers.

4. Botrytis blight or gray mold on euonymus liners.

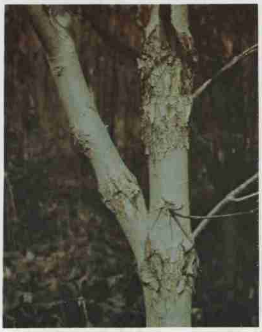
5. Mycelium and young southern stem blight sclerotia on ajuga.



6. Aucuba wilting from southern stem blight.

7. Mature sclerotia at base of aucuba stem.

COLOR PLATE VI



1. Dogwood canker.

2. Powdery mildew on Leucothoe plant.

3. Powdery mildew on underside of Leucothoe leaves.

4. Lesion nematode on American boxwood.

5. Web blight on Japanese holly.

6. Botrytis canker on rose cane.

7. Powdery mildew overwintering on rose cane.

8. Chamaedorea palm—*Glicladium vermoeseni* leaf and stem rot.

COLOR PLATE VII

1. Damping-off of seedlings.

2. Azalea leaf gall.

3. Camellia flower blight.

4. Black spot of rose.

5. Apple scab on fruit and leaf.

6. Powdery mildew of photinia.

7. Powdery mildew on apple.

8. Cedar-apple rust.

9. Dieback of camellia.

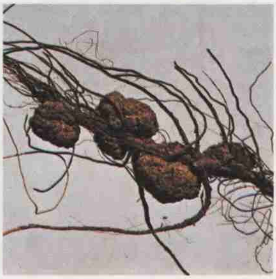
COLOR PLATE VIII

1. Powdery mildew.

2. Anthracnose on sycamore.

3. Anthracnose of dogwood.

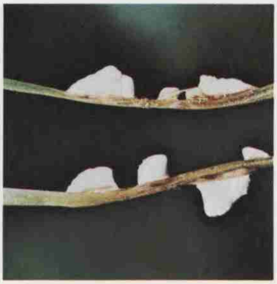
4. Oak leaf blister.



5. Crown gall on roots.

6. Fire blight.

7. Eastern gall rust of pine.



8. Pine needle rust.

9. Scab on pecan.

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