FAO/IPGRI Technical Guidelines for the Safe Movement of Germplasm No. 21

Pinus spp.

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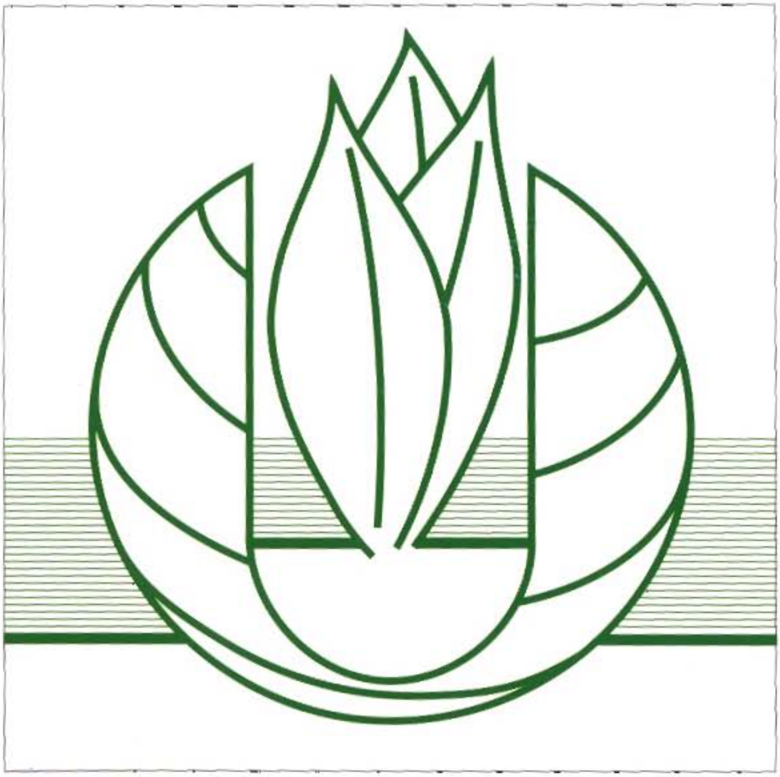

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Previously Published Technical Guidelines for the Safe Movement of Germplasm

These guidelines describe technical procedures that minimize the risk of pest introductions with movement of germplasm for research, crop improvement, plant breeding, exploration or conservation. The recommendations in these guidelines are intended for germplasm for research, conservation and basic plant breeding programmes. Recommendations for commercial consignments are not the objective of these guidelines.

Cocoa	1989
Edible Aroids	1989
Musa (1st edition)	1989
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INTRODUCTION

The collection, conservation and utilization of plant genetic resources and their global distribution are essential components of research activities underpinning the implementation of international crop and tree improvement programmes.

Inevitably, the movement of plant germplasm involves a risk of accidentally introducing associated plant pests.[1] Pathogens that remain symptomless in plants, such as viruses or bacteria, pose a special risk. To minimize such a risk, preventive measures and effective testing procedures are required to ensure that distributed material is free of pests of potential phytosanitary importance.

The international movement of plant germplasm for research (including plant biotechnology research), conservation and basic plant breeding purposes requires complete and current information concerning the phytosanitary status of the plant germplasm. In addition, the relevant and current national regulatory information governing the export and importation of the plant germplasm in the respective countries is essential. As the depository of the International Plant Protection Convention (IPPC), FAO collaborates with IPGRI to ensure and facilitate the safe movement of plant germplasm.

The IPPC is internationally recognized as the legal instrument and primary vehicle to achieve international cooperation in the protection of plant genetic resources from pests. The IPPC also seeks the harmonization and standardization of phytosanitary measures affecting international trade. IPGRI's mandate *inter alia* is to promote the collection, conservation and utilization of plant germplasm for the benefit of people throughout the world. The objective of the collaborative activities developed by IPGRI and FAO is to facilitate the safe movement of germplasm, for research purposes, by identifying technically sound practices that safeguard against the introduction and establishment of unwanted pests.

The purpose of the joint FAO/IPGRI programme is to generate a series of crop- or plant-specific technical guidelines that provide relevant technical information on pest recognition and detection procedures, to prevent the involuntary, international dissemination of pests of potential phytosanitary importance. The recommendations made in these guidelines are intended for small, specialized consignments used in research programmes, e.g. for collection, conservation and utilization for breeding of plant genetic resources.

1 The word 'pest' is used in this document as it is defined in the FAO Glossary of Phytosanitary Terms (1996): 'any species, strain or biotype of plant, animal, or pathogenic agent, injurious to plants or plant products'.

These technical guidelines are produced by panels of experts on the crop or plant concerned, selected in consultation with agricultural national and international research institutions working on the relevant crop group or plant species or genus. The experts contribute to the elaboration of the technical guidelines in their personal capacity and do not represent or commit the organizations for which they work. The guidelines are intended to provide the best possible phytosanitary information to institutions involved in small-scale plant germplasm exchange for research purposes. FAO, IPGRI and the contributing experts cannot be held responsible for any problems resulting from the use of the information contained in the technical guidelines. The technical guidelines reflect the consensus and knowledge of the specialists who attended the meeting, but the information provided needs to be regularly updated. The experts who contributed to the production of the technical guidelines are listed in this publication. Correspondence regarding this publication should be addressed to either IPGRI or FAO.

The guidelines are written in a concise style to keep the volume of the document to a minimum and to facilitate updating. Suggestions for further reading are provided, in addition to specific references cited in the text (mostly for geographical distribution, media and other specific information). The guidelines are divided into two parts. The first part makes general and technical recommendations on safe procedures to move *Pinus* spp. germplasm. The second part covers pests of phytosanitary concern for the international or regional movement of *Pinus* spp. Seed and seedling diseases are followed by foliage diseases, stem diseases, rusts and, lastly, pine wilt disease. The remainder of the publication is devoted to insects. The information given on a particular pest is not exhaustive but rather concentrates on those aspects that are most relevant to the safe movement of germplasm.

Because eradication of pathogens is extremely difficult, and even low levels of infection or infestation may result in the introduction of pathogens to new areas, no specific information on treatment is given in the pest descriptions. A pest risk analysis (PRA) will produce information on which management options are appropriate for the case in question. General precautions are given in the Technical Recommendations.

There are several features of pine species that make this genus of trees of particular concern with regard to movement of germplasm, namely:

- Pine species are among the most widely occuring trees in the Northern Hemisphere, where they are important components of boreal, temperate, sub-tropical and tropical forests. The natural range of the Asian species *P. merkusii* extends into the Southern Hemisphere.
- Pine species are highly valued for introduction and planting in many parts of the world because they are fast growing, easily cultivated and suitable for industrial plantations, agroforestry and community forestry. Pines supply many valuable products, including lumber, pulpwood, fuelwood, resin and edible nuts.

- At the end of 1990, there were an estimated 4.59 million ha of pine plantings in the tropics, comprising 10.5% of all tropical forest plantings (FAO 2000)[1]. Pine species are also a major plantation species in temperate regions of Asia, Europe and North America.
- Pine species have been widely planted in the Southern Hemisphere. A single pine species, *Pinus radiata*, has become an important plantation species in Argentina, Australia, Chile, New Zealand and South Africa. Chile and New Zealand each have in excess of 1.5 million ha of *P. radiata* plantations.
- Large quantities of seed are being collected in both native forests and plantations, to be distributed worldwide.
- The introduction of pine species into geographical areas where they are not indigenous has created additional pools of potentially susceptible hosts for the potential wider dissemination of pine pests.
- Many pests can naturally negatively affect the productivity of pine trees. In addition, there are several documented cases of accidental introductions of pests of pine on plant germplasm, which have caused devastating losses to both natural and planted forests. These cases, include the introduction of the white pine blister rust fungus, *Cronartium ribicola*, into North America; the pine woolly adelgid, *Pineus boerneri*, into Kenya and Zimbabwe; and, more recently, the loblolly pine scale, *Oracella acuta*, into China.

The present guidelines were developed at a meeting held in Opocno, Czech Republic, from 14 to 16 October 1996. The Forestry and Game Management Research Institute, Jiloviste-Strnady, Czech Republic, hosted the meeting. The participants in this meeting are listed below. We are grateful to Dr Charles S. Hodges from North Carolina State University for a critical review of the manuscript.

Guideline update

To be useful, the guidelines need to be updated when necessary. We ask our readers to kindly bring to our attention any developments that possibly justify a review of the guidelines, such as new records, new detection methods, or new control methods. For your convenience, please use the form provided on the last page of this publication.

1 FAO is presently updating all existing information in a Plantation Database (PDB 2002).

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GENERAL RECOMMENDATIONS

- Where possible, pest-risk analysis (PRA) should precede the movement of plant germplasm.
- Refer to the FAO (1996) publication International Standards for Phytosanitary Measures (ISPM) No. 2: *Guidelines for Pest Risk Analysis.*
- Plant germplasm should be obtained directly from the nearest source of healthy material consistent with technical objectives.
- Upon receipt, all material should be kept in isolation from other planting material, and planted under conditions conducive for symptom expression, with sufficient time allowed for potential pests to appear.
- Plant germplasm should undergo inspection and testing for presence of pests, and appropriate treatment applied as necessary or requested.
- Plant germplasm should not be released into the field unless it is confirmed to be pest-free. If not pest free, the germplasm should be treated to make it pest free, or destroyed together with the pests detected.
- Movement of seedlings, scions and other rooted material is **not recommended** because this material poses a high risk of transmitting pests.
- All packaging material used in the movement of germplasm should be destroyed.

TECHNICAL RECOMMENDATIONS

The selection of the method of plant germplasm transfer should meet the technical objectives of the germplasm use, e.g. conservation, evaluation, genetic improvement, etc.

1. Seed

- Always extract seeds from pine cones prior to shipment. Never ship pine cones containing seed because the cones may also harbour potentially damaging pests.
- Do not collect pine cones or seeds for germplasm exchange from the forest floor or animal caches.
- Seed storage facilities should be routinely sanitized using suitable/available disinfestants.
- Seed lots intended for storage or shipment should be free of debris, especially soil particles and remnants of pine cones and foliage. Seed should be air-dried to a moisture content recommended to maintain long term viability.
- If chemical treatment of seed is required prior to shipment, use materials and procedures that are currently approved and recommended for that purpose. It should be noted that seed treatment for pest control may not completely eradicate the target pest.
- Seed treatment with certain pesticides may affect seed viability. Should seed treatment with such pesticides be required, it should be done immediately prior to sowing. **Before** treatment of the seed, it is important to check to see whether the pesticides used are registered and/or accepted in the country of destination.
- Seeds should be germinated in a sterile substrate.

2. *In vitro* material

- *In vitro* material should be derived from healthy sources.
- *In vitro* material should be shipped in sealed, transparent containers and visually inspected before dispatch and immediately upon receipt at destination. Ideally, *in vitro* material should be indexed for the presence of systemic pathogens. Infected or contaminated material should be destroyed. When explants must be moved, they should be moved in a sterile medium.

3. Scions

- Where scion material is needed to meet germplasm management objectives, a thorough pest risk analysis (PRA) should be made prior to shipment. Appropriate pest management procedures suggested by the PRA analysis should be followed.

- Tools used for making cuttings (secateurs and other cutting tools) should be cleaned and surface disinfected between each cut, such as by dipping in a 0.5–1.0% sodium hypochlorite solution.
- Cuttings should be taken from trees that show no visible symptoms or signs of pest activity.
- If scions received at the final destination show visual or laboratory evidence of pest activity, they should be destroyed immediately.
- Any material not required for grafting should be destroyed.

4. Pollen

- Although some pine germplasm is moved as pollen, there is little information available on pests associated with pollen or practical experience on how to control potential pests associated with pollen. At the very least, the pollen should be examined by light microscopy for the presence of visible pests. Contaminated or infected pollen should be discarded.

INTERNATIONAL DISTRIBUTION OF GERMPLASM

- Movement of germplasm should comply with the regulatory requirements of both the exporting and receiving countries.
- A description of tests that have been performed to assess the health of the germplasm should accompany the shipment.
- If germplasm is re-exported, copies of the original documents should accompany the shipment, together with additional descriptions of any actions taken during transit that could affect the health or quality of the consignment.

DISEASES AND DESCRIPTIONS OF PATHOGENS

There are several diseases of global distribution that impede the successful cultivation of native and introduced pine species. However, some of the most serious losses have occurred when pine pathogens have been unintentionally moved to new localities, where they can be even more damaging than in their native habitat. Probably the best known example is white pine blister rust, *Cronartium ribicola*, which, after being introduced from its native Asia to Europe and then to North America, severely damaged valuable pine species in these areas. Of equal concern is the introduction of new strains of pathogens, such as the European race of *Scleroderris* canker, *Gremmeniella abietina*, introduced from Europe into eastern Canada and northeastern USA. In these American localities, the European race is considerably more virulent than the indigenous race of the pathogen. While attention often focuses on pathogens that have been moved internationally, domestic movement of pathogens within large countries is also a major concern, e.g. domestic movement of the root rot pathogen, *Phytophthora cinnamomi*, in Australia. Many pathogen introductions have occurred on seedlings, and it is for this reason that we have recommended that pine germplasm should never be moved in the form of seedlings.

While the above-mentioned diseases have gained worldwide notoriety, there are numerous other diseases that have been or could be moved on germplasm, especially on scions or seeds. One of the most insidious aspects of plant disease is that there is frequently a latency period between the time of infection and symptom appearance. With some rusts, for example, this period can take up to two years or more; consequently, infected, asymptomatic germplasm could unknowingly be transported. Another aspect of rust pathogens is that many of them complete part of their life cycle on one or more botanically unrelated alternate host plants. Hence, movement of alternate plant hosts for pine rusts should also be of concern to countries interested in preserving their pine forests.

An attempt has been made to mention the most important issues concerning the phytosanitary aspects of moving pine germplasm between different geographic locations. Limitations of space in this publication precludes fuller treatment, but many other useful recommendations and information on pine diseases can be found in more detailed publications listed in the general bibliography.

Seed and seedling diseases

Fusarium *seed and seedling diseases*

Causal organisms
The fungal genus *Fusarium* contains many species, and often *formae speciales* therein, that attack numerous hosts, including pine trees of all ages. Among the diseases caused by *Fusarium* spp. are root rots, wilt, blight and damping-off. Damping-off can be especially damaging on nursery-grown pine seedlings, where the prominent pathogens are *F. acuminatum, F. avenaceum, F. moniliforme, F. oxysporum* and *F. solani*, some of which can also induce root rots of older seedlings. Many *Fusarium* species are seed-, water- and soil-borne.

Hosts
Under optimum conditions, e.g. abundant fungus inoculum, high humidity and warm weather, susceptible pine species are likely to succumb to damping-off, as in the case of *Pinus banksiana, P. elliottii, P. palustris, P. resinosa, P. sylvestris* and *P. taeda* (Hwang *et al.* 1995; Huang and Kuhlman 1991; Uscuplic and Lazarev 1981; Pawuk 1978, 1979). Pine species susceptible to root rots include *P. patula, P. resinosa, P. strobus and P. taeda* (Juzwik and Rugg 1996; Farquhar and Peterson 1991; Rowan 1981). *P. taeda* is susceptible to top blight (Affeltranger 1983).

Geographical distribution
Fusarium species occur worldwide and probably under all (terrestrial and aquatic) conditions (Bloomberg 1981; Nelson *et al.* 1983). However, pathogenic interactions between some pine and *Fusarium* species under different environmental conditions may give rise to localized pathogenic variants or pathogen–host interactions, which merits the exclusion of exotic *Fusarium* spp. from pine germplasm.

Significance
Because (container) nurseries normally use pathogen-free growing media, seed and seedling losses associated with the presence of *Fusarium* spp. are normally minimal. However, seed and seedling losses in bareroot nurseries vary according to locations and years, i.e. from insignificant to almost total loss, depending upon numerous factors, particularly the abundance and kind of fusaria present, host susceptibility, weather, and soil factors such as pH and drainage.

Symptoms and signs
Fusarium species cause pre- and post-emergence damping-off (Fig. 1), root rots (Fig. 2) and foliage diseases of pine trees. In pre-emergence damping-off, seeds or germinating seedlings are rotted before emergence, thus no seedlings appear. In post-emergence damping-off, the young, non-woody stems of seedlings decay at ground level, resulting in seedling shoots toppling over and subsequent death. Root rot symptoms include

chlorosis, then desiccation and finally reddening of needles, and frequently the shoot tip becomes crosier-shaped. Shoots of affected plants remain upright and their roots decay. Symptoms of *Fusarium*-induced top blight often start at the growing tip of the plant, killing needles from the base upward. Under wet conditions the disease progresses laterally through the seedling canopy.

Biology and transmission
Fusarium spp. survive as resting or survival spores (chlamydospores) in pieces of organic matter, such as soil amendments, and recently killed host tissues, particularly root pieces. Host-produced chemicals often stimulate chlamydospore germination. The fungus penetrates adjacent seeds or young roots, which rot following their invasion by the fungus. Many fusaria also produce mycotoxins, some with phytotoxic effects. As the disease develops, other spores (macroconidia and sometimes microconidia) are produced to facilitate the propagation and dissemination of the fungus. Chlamydospores are normally produced under adverse conditions, specifically in response to lack of nutrients or to moisture stress. Movement by various means of contaminated soil, growing media, water and infected seed or seedlings contributes to local and long-distance dissemination of *Fusarium* spp. The occurrence of top blight is possibly associated with wind- and water-borne inocula, but little is known about the biology or pathogenicity of the fusaria involved, except that moist conditions favour the disease.

Fig. 1. (left). Pine seedling with symptoms of damping-off caused by *Fusarium* sp. (Dr J. Sutherland, Applied Forest Science, Victoria)

Fig. 2. (right). Typical symptoms of *Fusarium* root rot. (Dr J. Sutherland, Applied Forest Science, Victoria)

Detection

Fusaria causing damping-off, root rot and top blight can be readily isolated from affected plant parts by plating and incubating surface-sterilized tissues on culture media. Alternatively, unsterilized plant tissues can be put on *Fusarium*-selective media (James *et al.* 1991). The resulting cultures are identified using standard taxonomic keys, e.g. Nelson *et al.* (1983). Since spore characteristics are important in identification, simply placing diseased tissues in a humidity chamber to induce sporulation may suffice. Traps, such as pieces of host tissue, can also be placed in soil and the *Fusarium* subsequently isolated from them. Seeds suspected of harbouring fusaria could be plated directly onto selective media. Increasingly, molecular biology techniques are used for detecting and identifying *Fusarium* spp.

Seed or cold fungus

Causal organism
Caloscypha fulgens (Pers.) Boud.; anamorph: *Geniculodendron pyriforme* Salt.

Hosts
Seeds of many pine species, including *P. resinosa* and *P. contorta* (Egger and Paden 1986) and other conifers, especially spruce (*Picea* spp.) (Thomson *et al.* 1983).

Geographical distribution
The teleomorph is known from North America, the United Kingdom and Norway, indicating that the fungus may be present throughout the North Temperate Zone. In North America, the teleomorph releases ascospores between March and July, whereas cones for seed production are usually harvested in the autumn (Paden *et al.* 1978).

Significance
The pathogen was found to be seedborne in Canada and the USA. This seedborne fungus attacks and kills seeds under cool, moist conditions (particularly seeds on the ground), especially during stratification or after sowing in the nursery. As an inhabitant of forest duff, it may kill seeds during natural regeneration or following direct seeding. The fungus does not attack seedlings.

Symptoms and signs
The fungus often forms hard, whitish mycelial masses on seedcoats (Fig. 3). The hyphae are commonly present on seeds following stratification (Fig. 4). Seeds fail to germinate, their contents are mummified, but not rotted, and on cut seeds an indigo pigment may occur on the integument (Fig. 5). The teleomorph, producing

Fig. 3 (top). Whitish mycelial mass on seed coat of conifer seeds affected by the seed or cold fungus *Caloscypha fulgens*. (Dr J. Sutherland, Applied Forest Science, Victoria)

Fig. 4 (bottom). Mycelium of *Caloscypha fulgens* on conifer seeds following stratification. (Dr J. Sutherland, Applied Forest Science, Victoria) an apothecium with an orange hymenium, frequently occurs on forest duff shortly after spring snow melt, especially in mountain forests.

Biology and transmission

The fungus is a natural inhabitant of forest duff, thus seedlots most likely to be contaminated by the pathogen are those originating from ground-collected cones, such as from squirrel caches or cones picked from logging slash in contact with the forest floor. Seedlots from non-serotinous species (whose cones open when mature), e.g. *P. resinosa*, are more likely to be infected than are seedlots of serotinous species, such as *P. contorta*, which are seldom if ever infested. Even low levels of seedlot infestation are important, since the fungus spreads during cool, moist conditions, particularly during seed stratification. Fungus growth is best at 20°C, but can develop at temperatures as low as 5°C. However, dry seeds that are kept at low temperatures during long-term storage are in no danger. The fungus spreads between cones and seeds and mycelium penetrates seeds directly.

The role of ascospores and conidiospores in seed infection and long distance spread is unknown. It is known that squirrels and certain other forest rodents transport and eat infected cones and seeds (and perhaps *C. fulgens* fruiting bodies). They therefore have a role in localized dispersal.

Detection

Surface sterilize at least 500 seeds per seedlot with 30% hydrogen peroxide for 30 minutes and then plate the seeds onto 2% water agar in Petri dishes. Incubate for 2–3 weeks at 15–20°C, during which weekly observations with a stereomicroscope reveals the characteristic indigo-coloured, verrucose, right-angle-branching, hyphae of the fungus growing from diseased seeds (Fig. 6). Sutherland (1987) gives spore and cultural characteristics of the fungus.

Fig. 5 (top). Indigo-coloured mycelium of *Caloscypha fulgens* growing on water agar. (Dr J. Sutherland, Applied Forest Science, Victoria)

Fig. 6 (bottom). Mycelium of *Caloscypha fulgens* growing from an infected conifer seed. (Dr J. Sutherland, Applied Forest Science, Victoria)

Foliage diseases

Charcoal root rot

Causal organism
Macrophomina phaseolina (Tassi) Goid.

Hosts
Numerous plant species are attacked, including conifers. Most pine species are susceptible. *P. radiata* (Old 1981; Magnani 1979), *P. patula*, *P. elliottii* (De la Cruz and Hubbell 1975), *P. taeda* and *P. palustris* are severely affected in the USA (Barnard 1994). *P. pinaster*, *P. muricata*, *P. ponderosa*, *P. echinata*, *P. brutia* (Reuveni and Madar 1985; Khalisky *et al.* 1981), *P. caribaea* and *P. lambertiana* (Jamaluddin and Dadwal 1984) have also been reported as susceptible to *M. phaseolina*.

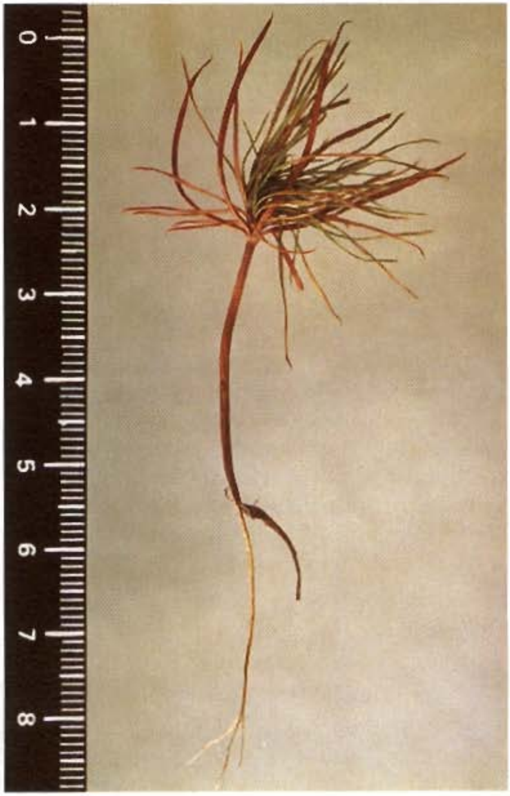

Fig. 7. Decayed roots and yellowing of needles following infection with *Macrophomina phaseolina*. (Dr H. Peredo, Universidad Austral de Chile, Valdivia)

Geographical distribution

Pantropical and subtropical. Some reports on pines include: North America (Barnard 1994; Smith 1969; Seymour and Cordell 1979); Israel (Reuveni and Madar 1985); India (Jamaluddin and Dadwal 1984); Iraq (Khalisy *et al.* 1981); Australia (Old 1981); and Italy (Magnani 1979).

Significance

Losses of nursery stock from premature plant death and culling may be high in heavily infested soil. Damage following planting of diseased nursery stock can be quite high, whereas losses caused by infection in plantations is usually low.

Symptoms and signs

Infection occurs through fine feeder roots and then progresses to larger roots, impairing water uptake. This process results in wilting and yellowing of foliage (Fig. 7). Infected roots decay, become covered with a dark-brown mycelial crust, and when advanced the bark sloughs off. Small, black sclerotia form in decayed roots, especially on the surface of the stele. Eventually the host dies, but it may survive for a prolonged period if infection is light.

Biology and transmission

The disease develops from sclerotia in nursery or plantation soil, leading to infection and subsequent root decay. The pathogen can be introduced into new areas on diseased seedlings. Sclerotia, produced on decayed roots, may remain dormant for many years and cause disease under favourable conditions. The pathogen has been detected on pine seeds (Dr J. Sharma, pers. comm.).

Detection

Symptoms and signs include wilting and yellowing of foliage, and black sclerotia on roots, respectively.

Diplodia *shoot blight and related diseases*

Causal organism

Sphaeropsis sapinea (Fr.) Dyko & Sutton, syn. *Diplodia pinea.*

Hosts

Over 33 *Pinus* spp., as well as other conifers, are susceptible: e.g. *Pinus banksiana* and *P. resinosa* (Blodgett *et al.* 1997a,b; Blodgett and Stanosz 1997; Stanosz *et al.* 1997); *P. strobus, P. sylvestris, P. edulis, P. mugo, P. nigra, P. ponderosa* (Stanosz *et al.* 1996); *P. eliottii* (Fraedrich *et al.* 1994); *P. radiata and P. patula, P. taeda, P. virginiana* (Swart *et al.* 1991).

Geographical distribution

Cosmopolitan. Reported on pines in North America: Great Lakes region (Blodgett *et al.* 1997); west-central USA (Stanosz *et al.* 1996); southern USA (Affeltranger 1981); Canada (Stanosz and Smith 1996); New Zealand, South Africa, Australia (Smith and Stanosz 1996; Swart and Wingfield 1991); and the Netherlands (Dijk *et al.* 1992).

Fig. 8. Symptoms of Diplodia blight and dark fruit bodies of the pathogen *Sphaeropsis sapinea* on a young, container-grown Ponderosa pine seedling (*Pinus ponderosa*). (Dr J. Sutherland, Applied Forest Science, Victoria)

Significance

S. sapinea is a destructive pine pathogen, particularly in New Zealand, Australia and South Africa (Chou 1976; Currie and Toes 1978; Swart *et al.* 1987). Its notoriety stems from the damage that it causes to adult pine trees following hail wounding.

Symptoms and signs

S. sapinea causes damping off and collar rot of seedlings, and attacks mature trees causing shoot blight, branch and bole canker, sap-stain and root disease (Fig. 8). Shoot blight is the most common symptom and occurs on both seedlings and mature trees. Infection causes dieback, and if it occurs over several consecutive years it results in stunting and deformation of trees, and ultimately death. Infection of terminal shoots is considered the most damaging form of shoot blight because it drastically affects the length of the marketable bole. Some symptoms are unique or rare in certain parts of the world or on certain species of *Pinus*. For example, *S. sapinea* causes a serious root disease only in South Africa (Wingfield and Knox-Davies 1980). In Uruguay, *Lasiodiplodia theobromae* and *Sphaeropsis sapinea* cause red top disease, whose main symptoms include top kill accompanied by copious amounts of resin and reddish-brown needles.

Biology and transmission

S. sapinea exists as a saprophyte on healthy and dead cones, twigs and needles. It has recently been found as an endophyte within apparently healthy pine cones (Smith *et al.* 1996). It is an opportunistic pathogen, causing disease when trees are wounded, e.g. by hail, or stressed by factors such as drought. Disease outbreaks in Uruguay have been associated with wounding by the European pine shoot moth, *Rhyacionia buoliana*. However, pine shoots are also susceptible to infection under optimum climatic conditions in the absence of hail and drought (Swart *et al.* 1987 1988). Moisture is necessary for infection, and young shoots become infected when rain coincides with warm temperatures at the onset of growth. *S. sapinea* is also seed-transmitted (Rees and Webber 1988).

Detection

Conidia are produced on infected tissue incubated in a moist chamber. The pathogen can be isolated directly from diseased tissues or from seeds plated onto standard culture media (Uzunovic *et al.* 1996).

Modern molecular techniques are also available to characterize *S. sapinea* isolates (Stanosz *et al.* 1996). There is a selective medium for *S. sapinea* (Swart *et al.* 1987).

Lophodermium *needle cast*

Causal organism

Lophodermium spp., particularly *L. seditiosum* Minter, Staley & Millar [anamorph: *Leptostroma rostrupii* Minter], but also *L. pinastri* (Schrad.) Chev. and, to a lesser extent, *L. baculiferum*.

Hosts

Pinus spp., such as *P. sylvestris* (Kowalski 1990); *P. radiata* (Choi and Simpson 1991); *P. ponderosa* (Hoff 1988); *P. resinosa* (Byther and Davidson 1979); *P. caribaea* (Hong 1977); *P. strobus* (Morgan-Jones and Hulton 1977); *P. pinaster* (Bega *et al.* 1978); *P. densiflora* Nicholls and Ostry 1978); and *P. banksiana* (Carter 1975).

Geographical distribution

Cosmopolitan in temperate zones, e.g. Poland (Kowalski 1990); New South Wales (Choi and Simpson 1991); Wisconsin (Ostry and Nicholls 1989); Socialist Republic of Yugoslavia (Lazarev 1986); Montana (Hagle and Kissinger 1986); Latvian ex-SSSR (Kotov and Kotova 1981); Russia (Aminev 1980); the Netherlands (van Leven 1979); Finland (Kurkela 1979); Canada (Minter 1980); Malaysia (Hong 1977); Hungary (Pagoni 1977); Hawaii (Bega *et al.* 1978), Turkey (Oymen 1975); and Sweden (Martinsson 1975) (some of these references include other *Lophodermium* spp.). For *L. pinastri* in the USA, the states of Michigan, Minnesota, Wisconsin, Washington, Indiana, Ohio, West Virginia, Pennsylvania, Vermont, Connecticut and North Carolina have been reported as affected by this disease (Carter 1975).

Fig. 9. Black ellipsoid ascocarps of *Lophodermium pinastri* on a Scots pine needle. The dark perimeter line around the ascocarps and the black zone line are characteristic. (Dr M. Svecova, Prirodovedecka Fakulta UK, Prague)

Significance

L. seditiosum is an important pathogen of nursery pine trees in temperate regions. Many other species occur as saprophytes in the tropics and subtropics. The other species also seem to be pathogenic, sometimes in the tropics, often in temperate regions.

Symptoms and signs

Spring 'reddening' in forest nurseries, leaf spotting and severe needle cast, sometimes leading to the death of trees in older pine plantations. Most infections occur on needles on the lower half of affected pines. Signs include black, oval, elongated fruiting bodies on infected needles.

Biology and transmission

L. seditiosum inhabits primary and secondary needles. Ascocarps develop on both attached and fallen needles, where they mature by late summer and sporulate during rainy weather, being forcibly discharged and then carried by the wind.

Detection

The ascocarps are very conspicuous (Fig. 9). Cultures of the various isolates differ in physiological characters and colour (white, fawn, tan, yellow, orange or various shades of brown).

Mycosphaerella diseases

Brown needle blight

Causal organism

Mycosphaerella gibsonii H. Evans, syn. *Cercospora pini-densiflorae*; anamorph *Pseudocercospora pini-densiflorae* (Hori & Nambu) Deighton.

Hosts

The disease affects numerous *Pinus* spp under natural conditions. *P. roxburghii* (Sujan-Singh *et al.* 1988); *P. kesiya* (de la Cruz *et al.* 1984); *P. massoniana* (Uhlig 1977); *P. merkusii* (Kobayashi et al. 1979.); *P. thunbergii* (Ono 1972); *P. radiata; P. caribaea, P. oocarpa, P. strobus* and *P. patula* (Sujan-Singh and Khan 1988). Other conifers, such as *Abies veitchii, A. sachalinensis, Cedrus deodara, Picea glehnii, P. jezoensis, Pseudotsuga menziesii* and *Larix leptolepis,* were shown to be susceptible after artificial inoculation. *Pinus kesiya, P. elliottii* and *P. clausa* have been reported as resistant to the pathogen in India (Sujan-Singh and Khan 1988).

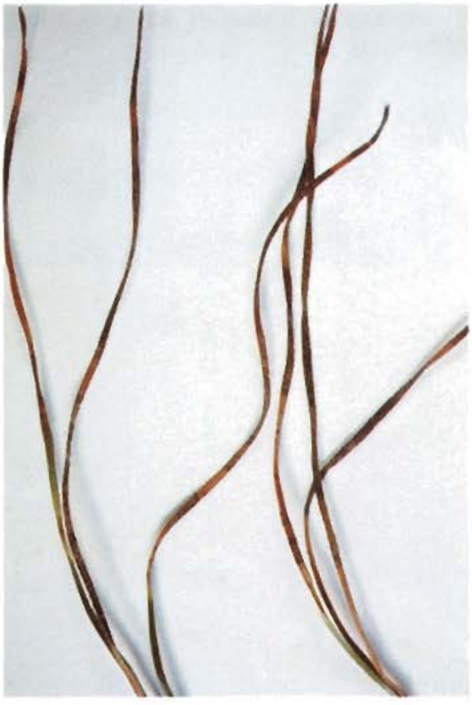

Fig. 10. Brown needle disease caused *by Mycosphaerella gibsonii:* Yellow-brown lesions alternate with greyish needle bands. (Dr Y. Suto, Shimane Prefecture Forest Research Centre, Yatsuka-gun)

Geographical distribution
Occurs throughout much of Africa, the Caribbean, Central America and Asia. The teleomorph occurs in some localities in Africa and Asia: India (Sujan-Singh *et al.* 1988); Philippines (de la Cruz *et al.* 1984); Japan (Okamoto *et al.* 1988; Suto 1982); Nepal (Ivory 1985); South Africa (Ivory and Wingfield 1986); Central America (Evans 1984) and the Caribbean (Ivory 1994); Bangladesh, Madagascar, Papua New Guinea, Swaziland, Thailand and Zambia (Ivory 1994).

Significance
Disease severity varies according to the pine species attacked, age of the tree at infection time, and environmental conditions.

Symptoms and signs
Lesions are 5–10 mm long, initially yellow, then turn yellow-brown. On blighted needles these lesions, with dark, minute fruit bodies, alternate with greyish needle bands (Fig. 10). Stroma of the fungus erupts through stomata, and, under humid conditions, dark olive tufts of conidia develop on the stromata. The disease gradually progresses upward from the lower needles on seedlings and trees.

Biology and transmission
The disease spreads to new areas on infected nursery stock. Hyphae overwinter in affected needles, or sometimes as latent infections in sound needles. Conidia are dispersed by rain splash. Ascomata are sometimes produced in stromata, but the role of ascospores in development of epidemics is unknown. In culture, the fungus grows slowly to produce dark, compact, olive-grey colonies, which if exposed to black light produce conidia (Kiyohara and Tokushige 1969).

Detection
The presence of needle blight and characteristic fruiting bodies.

Brown spot needle blight

Causal organism
Mycosphaerella dearnessii Barr; syn. *Scirrhia acicola*; anamorph: *Lecanosticta acicola* (Thüm.) Syd., syn. *Septoria acicola*.

Hosts
Many *Pinus* spp., including *Pinus taeda, P. elliottii, P. thunbergii, P. palustris, P. echinata, P. caribaea, P. clausa* (Li *et al.* 1986); *P. radiata, P. patula* (Ramirez 1981) *P. mugo* (Cech 1997); *P. uncinata* (Holdenrieder and Sieber 1995). *Pinus massoniana, P. taiwanensis* and *P. fenzeliana* have been reported as resistant to the pathogen (Li *et al.* 1986).

Geographical distribution
Widespread in the Americas: USA (Huang *et al.* 1995), Central America (Evans 1984); also in China (Li *et al.* 1986), France (Levy and Lafaurie 1994), Germany (Pehl 1995),

Fig. 11. Brown spot needle blight caused by *Mycosphaerella dearnessii*: light-brown lesions of about 3 mm length. (Dr Y. Suto, Shimane Prefecture Forest Research Centre, Yatsuka-gun)

Spain, Switzerland (Holdenrieder and Sieber 1995); Austria (Cech 1997) and the former Yugoslavia; Colombia (Ramirez 1981); and Cuba (Carreras *et al.* 1989).

Significance
The disease is most serious on *Pinus palustris* where it stunts and kills seedlings and saplings in the 'grass' stage. It also attacks other pines, resulting in seedling death and slow growth of plantation trees. *P. ponderosa* and *P. sylvestris* Christmas trees suffer browning and defoliation.

Symptoms and signs
There are two types of lesions, each about 3 mm long, with the most common being small, greyish-green spots that later become straw-yellow and then light brown (Fig. 11). Another type of lesion encircles the needle and is an amber-yellow band, with a small brown centre. Under high humidity, conidia exude from the stromata in white to dark-green, wedge-shaped tendrils.

Biology and transmission
Long-distance spread of the pathogen occurs on diseased nursery stock, while local spread, e.g. within nurseries, is by rain-splashed conidia. Ascomata form mainly on dead, fallen needles. Ascospores, probably air-borne, probably account for medium-distance spread. The fungus grows very slowly in culture, forming pink-grey, brown or greenish-black stromatic colonies, with slimy masses of pink-grey or greenish conidia.

Detection
Presence of needle blight and characteristic fruiting bodies.

Red band needle blight

Causal organism
Mycosphaerella pini Rostr., syn. *Scirrhia pini;* anamorph: *Dothistroma septospora* (Dorog.) Morelet, syn. *D. pini.*

Hosts
Many *Pinus* spp., including *Pinus nigra* (Vidakovic *et al.* 1986; Butin 1986); *P. pinea* (Neves *et al.* 1986); *P. wallichiana* (Zakaullah *et al.* 1987); *P. caribaea* (Foster 1982); *P. radiata* (Lambert 1986; Marks and Hepworth 1986); *P. cembra, P. aristata, P. koraiensis, P. tabuliformis* (Lang and Karadzic 1987); *P. canariensis* (Roux 1984); *P. mugo* (Pehl and Butin 1992); *P. thunbergii* (Suto 1990); *P. ponderosa* (Eldridge *et al.* 1980) and other conifers, including *Picea abies, Pseudotsuga menziesii* and *Larix decidua. P. sylvestris* and *P. densiflora* have been reported as sources of resistance (Vidakovic *et al.* 1986; Lang and Karadzic 1987).

Geographical distribution
Widespread in Africa: South Africa (Roux 1984); the Americas: Jamaica (Foster 1982), Central America (Evans 1984), Ecuador (Galloway 1987), Canada (Hunt 1995); Asia: Pakistan (Zakaullah *et al.* 1987), Japan (Suto 1990); Europe: Croatia (Vidakovic *et al.* 1986), Azores and Portugal (Neves *et al.* 1986), Germany (Butin 1986; Lang and Karadzic 1987), Spain (Cobos-Suarez and Ruiz-Urrestarazu 1990), Hungary (Koltay 1997); Australia (Lambert 1986; Marks and Hepworth 1986) and New Zealand (Ray and Vanner 1988; Bulman 1993).

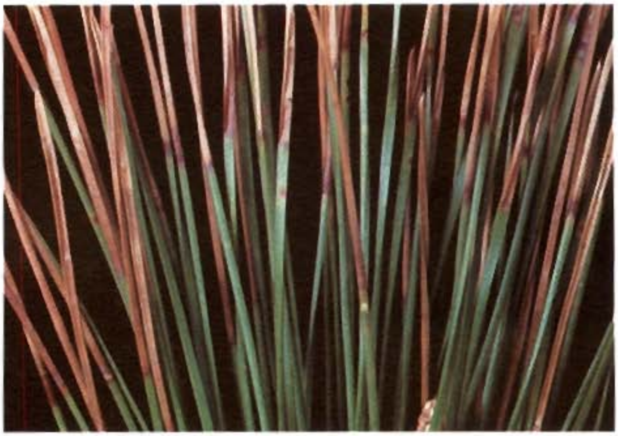

Fig. 12. Red band needle blight: Necrotic tips of needles infected with *Mycosphaerella pini.* (Dr Y. Suto, Shimane Prefecture Forest Research Centre, Yatsuka-gun)

Significance
The disease causes premature needle drop and subsequent growth reductions in *Pinus* spp. Severe infection kills trees. *P. radiata* is highly susceptible to the disease, which has prevented establishment of *P. radiata* plantations in several African countries. A serious disease of *P. radiata* in areas with summer rainfall.

Symptoms and signs
Initially, lesions appear as chlorotic bands, then turn brown to reddish-brown, and finally necrotic. The tips of infected needles above the lesion frequently wilt, turn brown, then necrotic (Fig. 12). Small, black stromata of the fungus, bearing conidiomata and occasionally ascomata, form in the needle cortex of the bands and emerge through epidermal slits. Under humid conditions, conidia ooze from the stromata in white to pink, wedge-shaped cirri.

Biology and transmission
Infected nursery stock accounts for long-distance spread, whereas local spread is associated with conidia in wind-blown water droplets. Ascomata form mainly on dead needles, either on the tree or after they are cast. Ascospores are probably air-borne and have a dispersal function. The fungus grows very slowly in culture, producing a red, water-soluble pigment. Cultures are pink-grey-brown with black stromata and produce masses of slimy pink, grey or greenish conidia.

Detection
Presence of needle blight and characteristic fruiting bodies.

Sirococcus *blight*

Causal organism
Sirococcus conigenus (DC.) Cannon & Minter. There are many synonyms, including the most recently used ones of *S. strobilinus* and *Ascochyta piniperda*.

Hosts
S. conigenus affects numerous conifers, including many pine species: *Pinus resinosa* (Magasi 1991); *P. ponderosa, P. contorta* (Hamelin and Sutherland 1991); *P. cembra, P. mugo* (Schnell 1987); *P. strobus* (Campbell and Schlarbaum 1992); and *P. sylvestris*.

Geographical distribution
Sinclair *et al.* (1993) report the fungus as occurring on conifers in North America: USA (Campbell and Sclarbaum 1992); Canada (Magasi 1991; Hamelin and Sutherland 1991); California (Smith 1973); Europe: Germany (Schmidt 1997); Switzerland (Schnell 1987); and Spain (Muñoz-Lopez 1997).

Significance
This fungus, which can be seedborne on conifer seeds, including some pine species, causes a shoot blight or tip blight of numerous pine species and other conifers in nurseries, plantations and natural forests.

Symptoms and signs
The fungus causes tip-dieback, and branch and stem cankers on the current year's growth of trees of all ages. Resin is often exuded at the site of infection on new needles or current-year stems. Distal foliage droops, becomes yellow or reddish-brown, and, on young

Fig. 13. Lodgepole pine seedlings (two on right) with symptoms of *Sirococcus* blight (two on right; healthy seedlings, left). (Dr J. Sutherland, Applied Forest Science, Victoria)

seedlings, affected needles often die from the base upward. Elongated, sunken, purplish cankers may develop at the infection site, which restricts stem growth and frequently results in deformation of the shoot tip. Pycnidia of the pathogen normally form on killed tissue. Very young nursery seedlings may be either killed or severely deformed (Fig. 13). Damage on older trees may be confined to lower branches or result in severe defoliation, depending upon the host and long-term weather. In forest stands, small, light-stressed trees beneath larger, diseased trees may suffer most from the disease. Pycnidia are common on cones (Fig. 14), where seeds become infected.

Biology and transmission

Only the asexual stage of the fungus is known. Pycnidia, which produce pycnidiospores, are common on old cones, diseased foliage, and twigs. The disease cycle is completed in one year, but pycnidiospores may be released for up to a year thereafter. Although infection occurs on young needles and stems, only old (not current year) cones appear to be infected. Seedborne inoculum results in initial disease in the nursery, particularly on container-grown seedlings, from which the pathogen spreads and intensifies. In forest stands, and sometimes in nurseries, inoculum originates from diseased needles, twigs and old cones. Infection occurs as for nursery seedlings. Free moisture and temperatures of 10–25°C favour infection. Subsequent spread occurs throughout the growing season by wind- and water-borne pycnidiospores.

Detection

Seedborne inoculum can be detected by surface sterilizing seeds (the number of test seeds varies with the degree of accuracy required) with a surface sterilant, such as sodium hypochlorite or hydrogen peroxide and then plating them on malt agar. Following incubation at 20°C (8 hours light), the fungus, which grows from diseased seeds and sporulates on the germinants, can be detected using a stereomicroscope. A very sensitive and accurate detection method for seeds is to use the monoclonal antibody protocol developed by Mitchell (1988). The fungus is normally isolated from plant parts following surface sterilization and plating on a culture medium such as PDA, or the fungus is induced to sporulate by incubating unsterilized, diseased tissues in a humidity chamber.

Fig. 14. Pycnidia of *Sirococcus conigenus* on pine cone scales. (Dr J. Sutherland, Applied Forest Science, Victoria)

Stem diseases

Pitch canker

Causal organism
Fusarium subglutinans (Wollenw. & Reink.) Nelson, Tousson & Marasas f.sp. *pini* Carroll *et al.* (also referred to as: FSP or F.s. pini), syn. *F. moniliforme* var. *subglutinans*.

Hosts
Many pine species, such as *Pinus radiata* (Hoover *et al.* 1996); *P. taeda* (Carey and Kelly 1994); *P. patula* (Viljoen *et al.* 1995); *P. elliottii, P. canariensis, P. halepensis* (Correll *et al.* 1992); *P. serotina* (Kuhlman and Kade 1985); *P. virginiana; P. echinata; P. strobus* (Dwinell *et al.* 1985) and *P. palustris* (Runion and Bruck 1988) have been found to be naturally infected with pitch canker, while others are susceptible to artificial inoculations (McCain *et al.* 1987).

Geographical distribution
Pitch canker is native to the USA (McCain *et al.* 1987), particularly in southeastern USA (Dwinell *et al.* 1985; Runion and Bruck 1988), but it severely attacks pines in California (Correll *et al.* 1992), particularly the Monterey pine, which is on the verge of extinction due to this disease (McCain *et al.* 1987); it has also been reported from Canada (Hoover *et al.* 1996), Haiti (Hepting and Roth 1953), Japan (Kobayashi and Muramuto 1989), Mexico (Santos and Tavor 1991) and South Africa (Viljoen *et al.* 1994).

Fig. 15 (left). Pitch canker symptom on the trunk of a pine tree. (Dr T. Coutinho, University of the Free State, Bloemfontein)

Fig. 16 (right). Pitch soaked area around a canker. (Dr T. Coutinho, University of the Free State, Bloemfontein)

Significance

Pitch canker is a serious threat in nurseries, seed orchards, plantations and natural stands. Since this pathogen thrives under a wide range of environmental conditions, it is important to prevent its spread into new geographical areas.

Symptoms and signs

The pathogen infects both vegetative and reproductive structures of pines at any stage of their development. The characteristic symptoms are resinous, slightly depressed cankers on the trunk or large branches (Fig. 15) and shoot dieback in the upper crown. Large amounts of pitch accumulate on and below the cankers. Wood beneath cankers is deeply pitch soaked (Fig. 16), often to the pith. No swellings or callus develop on or around the canker. These characteristics distinguish pitch canker from other canker diseases. This pathogen also infects cones, which tend to be deformed and smaller than normal (Barrows-Broaddus 1987) and can cause a severe and extensive root disease of seedlings (Blakeslee *et al.* 1981; Viljoen *et al.* 1994).

Biology and transmission

The pathogen is opportunistic, relying on wounds for infection. Insects such as *Ips* spp., *Pityophthorus* spp., *Pissodes* spp. and *Conophthorus* spp. are reportedly associated with pitch canker. Conidia are air-borne and maximum dispersal occurs during precipitation and turbulent air conditions. The fungus is also soil-, water- and seedborne.

Detection

In the case of root disease, direct isolation on standard and selective culture media is an effective means of detection. Imported seeds should always be assayed for pitch canker. The distinguishing characteristics of the fungus are production of microconidia in false heads on polyphialides (Fig. 17) and absence of chlamydospores.

Fig. 17. *Fusarium subglutinans* f. sp. *pini*: production of microconidia in false heads on polyphialides. (Dr T. Coutinho, University of the Free State, Bloemfontein)

Scleroderris canker and shoot blight

Causal organism

Gremmeniella abietina (Lagerb.) Morelet; syn. *Scleroderris lagerbergii, Crumenula abietina;* anamorph: *Brunchorstia pinea* (P. Karst.) v. Höhn. By fatty acid and sterol profiles, as well as DNA-methods, it has been demonstrated that *G. abietina* comprises two varieties and *G. abietina* var. *abietina* has three races, one North American, one European and one Japanese (Asian race) (Hamelin and Rail 1997). The race concept has also been discussed by other workers (Hamelin *et al.* 1993; Müller *et al.* 1994).

Hosts

In Europe, *P. sylvestris* (Hansson 1996; Ranta and Neuvonen 1994; Kurkela 1983) and *P. nigra* (Stephan *et al.* 1984) are the main hosts. The North American race occurs mainly on *Pinus banksiana* and *P. contorta* (Hamelin and Rail 1997), but *P. resinosa* and *P. strobus* are also susceptible (Anderson and Mosher 1975; Karlman *et al.* 1994; Martinsson 1984; Laflamme *et al.* 1996). Other susceptible pine species include *P. cembra* (Donaubauer 1984), *P. nigra* (Rosnev and Petkov 1990), *P. rigida, P. mugo P. wallichiana* and *P. pinea* (Petrini *et al.* 1990). Some *Picea, Larix, Pseudotsuga,* and *Abies* species may also be affected (Laflamme *et al.* 1996; Skilling *et al.* 1984).

Geographical distribution

On *Pinus* spp., the disease occurs in the northcentral and northeastern USA (O'Brien and Miller-Weeks 1982; Skilling *et al.* 1984), in Ontario (Dorworth and Davis 1983), Quebec (Lachance 1984) and the Maritime Provinces (Magasi 1984) of Canada, and in northern Europe: e.g. Sweden (Hansson 1996; Petrini *et al.* 1990), Finland (Kallio *et al.* 1985; Capretti 1984), Bulgaria (Rosnev and Petkov 1990), Norway (Solbraa and Brunvatne 1994), and Austria (Breitenbach-Dorfer and Cech 1996). In Japan, the Asian race affects *Abies sachaliensis* (Hamelin and Rail 1997).

Significance

The fungus kills pine seedlings and transplants in nurseries, and damages plantation trees and older pine stands.

Symptoms and signs

On nursery seedlings, the first signs of the disease appear after spring snow melt and include drooping and very loosely attached needles at the shoot terminal. Later the apical buds and shoot bark become necrotic (Fig. 18). With nursery seedlings, discoloration begins at the needle base while the distal part remains green. Shoot damage on saplings and older trees may occur on the lateral branches and upper crown. In the forest, stem and branch cankers are common on affected pines (Fig. 19). The fungus causes a persistent green discoloration of the wood of cankered tissues and infected shoots.

Fig. 18 (top left). Infection with *Gremmeniella abietina* on Scots pine shoot in early spring. The needles are symptomless, the colonized bark tissue is turning brown. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Fig. 19 (top right). A canker caused by *Gremmeniella abietina*. Wood surface in the canker is typically yellowish-green. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Fig. 20 (right). Pycnidia of *Gremmeniella abietina* in a one-year-old Scots pine seedling. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Biology and transmission

Conidia or ascospores infect bracts of short shoots or bud scales. The pathogen remains latent until the next spring, when the first symptoms appear and conidia production begins in pycnidia (Fig. 20). Conidia are mainly spread by splashing rain, while ascospores, produced from midsummer to autumn, in apothecia (Fig. 21), are air-borne. The disease can be readily spread by infected, symptomless nursery seedlings.

Detection

Conidia are spindle-shaped, hyaline, slightly curved and 5-celled (common European or lowland race) or mostly 8-celled (alpine or northern race). The fungus can be readily isolated on malt extract agar, where the mycelium has a greenish or brownish cast. Typical conidia are formed on specifically enriched media (Uotila 1983).

Fig. 21. Apothecia of *Gremmeniella abietina* on the bark of Scots pine. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Terminal crook disease

Causal organism
Colletotrichum acutatum Simmonds f. sp. *pinea* Dingley & Gilmour.

Hosts
Pinus contorta, P. elliottii, P. pinaster and *P. radiata* (Nair *et al.* 1983; Peredo *et al.* 1979).

Geographical distribution
Australia (Anonymous 1967), Chile (Peredo *et al.* 1979), Kenya (Gibson and Munga 1969), and New Zealand (Nair and Corbin 1981; Vanner and Gilmour 1973).

Significance
Usually a nursery disease. Symptomless plants may carry the pathogen to plantations, where the disease develops and seedlings do not compete against weeds.

Symptoms and signs
C. acutatum affects the terminal bud of nursery seedlings. Diseased tips become crosier-shaped, affected tissues turn pink and the stem usually thickens below the lesion. Diseased seedlings are stunted, often less than half normal size, thickset, very rigid, and have numerous lateral shoots. Under moist conditions, viscous masses of salmon-orange spores develop on killed stems and needles.

Biology and transmission
Primary inoculum for infection of seedlings is soilborne, surviving as dark hyphae and chlamydospores on plant debris, and under favourable conditions producing new infections the following year. Seedlings older than 9–10 months are resistant. Sporulation and growth *in vitro* occur at 17–28°C and 8–31°C, respectively. Warm temperature favours infection.

Detection
The pathogen sporulates readily on potato dextrose agar (PDA), producing a carmine pigment and brightly coloured sporodochia. Conidia are typically cylindrical to fusiform, 9.5–15 µm × 3–4 µm in size. Conidiophores usually form sporodochia or acervuli with or without brown setae 25–64 µm long. Under moist conditions, viscous masses of salmon-orange spores form on killed stems and needles.

Pine rusts

Pine twist rust

Causal organism

Melampsora pinitorqua (A. Braun) Rostr. causes pine twist rust. This fungus belongs to a complex species called *M. populnea* P. Karst.

Hosts

Pinus sylvestris, P. pinea and *P. pinaster* are the most susceptible species, but *P. halepensis, P. mugo* and *P. nigra* are also susceptible (Longo *et al.* 1975). *P. banksiana* and *P. contorta* seem to possess some resistance to the fungus (Longo *et al.* 1980). Alternate hosts for *M. populnea* include aspen (Leuce group of *Populus*), and in Europe, usually *Populus tremula* L.

Geographical distribution

On *Pinus* spp., the disease has been recorded throughout Europe (Longo *et al.* 1980), and in the western Asiatic parts of the former USSR (Krutov 1981). The occurrence of *M. populnea* in South Africa and South America shown in map no. 389 (CMI 1972) are restricted to alternate hosts.

Fig. 22. Twist rust on Scots pine seedlings. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Significance
M. pinitorqua injures or kills elongating pine shoots in young stands. Serious infections decrease the technical value of trees due to crooked stems and repeated leader changes.

Symptoms and signs
The first symptom of infection is the appearance of narrow yellow spots, up to 1 cm in length, on the surface of elongating succulent shoots in early summer. Spermogonia develop on these spots, and they are followed by aecia in a few days. Aecia produce an orange-coloured mass of aeciospores. Early infection usually causes twisting or dying of shoots (Fig. 22). Late infection with aecial development causes only wounding of shoots. During the following winter, damaged shoots may break under the snow load.

Biology and transmission
The fungus overwinters in the telial state on aspen leaf litter. Basidiospores (Fig. 23) are dispersed during moist weather at the time of pine shoot elongation. Succulent pine shoots may be symptomless for up to one week after infection. During that week, the disease can be transmitted in scion material. Aecia develop on the infected shoot in 10–14 days. Aeciospores disperse on aspen leaves, on which several uredial cycles may develop during the summer. Aeciospores and urediniospores disperse in dry conditions. The fungus assumes the telial state with the onset of autumn (Kurkela 1973).

Detection
Infection on succulent shoots can be detected when yellow lesions with spermogonia appear on the shoot surface.

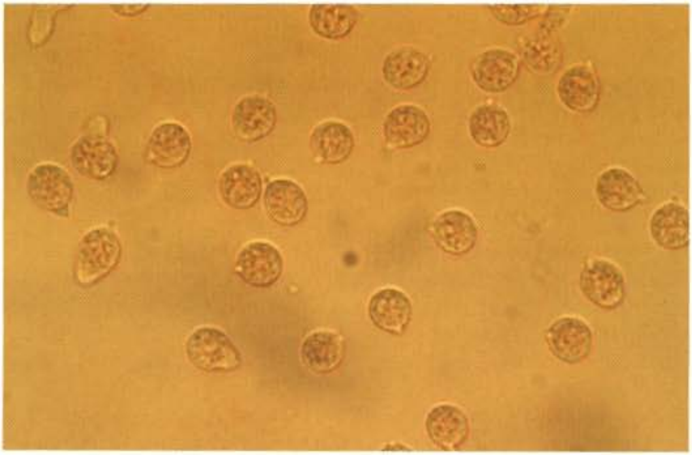

Fig. 23. Basidiospores of *Melampsora pinitorqua*. (Dr T. Kurkela, Finnish Forest Research Institute, Vantaa)

Stem and needle rusts

Causal organisms
There are about 15 species of *Cronartium* and four of *Endocronartium* causing the most damaging stem rusts of pine in the world. Pine needle rusts are caused by *Coleosporium* spp. Species of *Peridermium* are aecial forms of *Cronartium* spp., with the exception of *P. bethelii* Hedgec. & Long and a part of the *P. filamentosum* complex (Hiratsuka 1995).

Hosts and Geographical distribution
The details on hosts and geographical distribution of the most important rusts are given in Appendix 1. In general terms, stem rusts are caused by fungi of the genera *Cronartium* and *Endocronartium* (anamorph *Peridermium*), except for pine twist rust, which is caused by *Melampsora pinitorqua*.

White pine blister rust, *Cronartium ribicola*, is a native to Asia and was introduced into Europe and North America (APS 1997). In Asia, it has been reported from Pakistan attacking *Pinus wallichiana* (Zakaullaii 1994) and China, on *P. koraiensis* (Cheng-Dongsheng *et al.* 1998) and *P. takahasii* (Xue-Yu 1995). In Europe, *P. strobus, P. cembra, P. monticola* and *P. wallichiana* are infected in Rumania (Blada 1989; Borlea 1992). In Poland, *P. cembra, P. rigida* and *P. banksiana* showed different levels of susceptibility (Janczak 1997). The fungus is also important in Finland, Sweden and Italy (Kasanen 1997). In North America, C. ribicola attacks *Pinus lambertiana* in California (Kinloch and Dulitz 1990), *P. albicaulis* in northern Idaho (Tomback *et al.* 1995) and western Montana (Keane and Arno 1993) and *P. flexilis* in North Dakota (Draper and Walla 1993). In Canada, white pine blister rust has been reported to affect *Pinus strobus* (Lavallee 1992) and *P. monticola* (Hunt 1994).

Pinyon blister rust is caused by *Cronartium occidentale* and affects mostly *Pinus edulis* and *P. monophylla* in southwestern USA (APS 1997).

Fusiform rust, caused by *Cronartium quercuum* f. sp. *fusiforme*, attacks primarily *Pinus taeda* and *P. elliottii* in southeastern USA (APS 1997; Powers *et al.* 1993). *Pinus clausa* and *P. virginiana* have been reported to be susceptible to the (proposed) f. sp. *virginianae* of *C. quercuum* in the USA (Powers *et al.* 1991). *Cronartium quercuum* f. sp. *banksiana* causes the pine (*Pinus banksiana*) disease known as 'Eastern gall rust' in eastern North America (APS 1997).

Western gall rust of *Pinus banksiana, P. contorta, P. ponderosa, P. muricata, P. radiata, P. mugo, P. nigra, P. pinaster* and *P. sylvestris* occurs across northern North America and south to Virginia in the east, and to northern Mexico in the west. This disease is caused by *Endocronartium harnessii* (anamorph: *Peridermium harknessii*) (APS 1997). In Canada, Western gall rust has been observed in Pinus contorta (Kamp 1994) and P. banksiana (Hills *et al.* 1994).

The 'resin top' disease is another type of rust caused by *Cronartium flaccidum* (monocyclic rust *Peridermium pini*, also known as *Endocronartium pini*). This disease affects *Pinus halepen-*

sis, P. mugo, P. nigra, P. pinaster, P. pinea and *P. sylvestris* throughout Europe (APS 1997). The disease has been reported from Russia (Fedulov 1992), Finland (Pappinen and Weissenberg 1994), Scotland (Greig and Sharpe 1991), Italy (Moricca and Ragazzi 1996) and Germany (Majunke *et al.* 1997), mostly affecting *P. sylvestris.*

Comandra blister rust, caused by *Cronartium comandrae*, infects *Pinus banksiana, P. contorta* and *P. ponderosa* in the USA and Canada (APS 1997; Walla 1992; Karlman *et al.* 1997).

The 'stalactiform blister' rust pathogen, *Cronartium coleosporioides* (anamorph: *Peridermium stalactiforme*) infects mostly *Pinus contorta* and *P. banksiana* in North America, including Alaska and western North America (APS 1997).

The 'sweetfern blister' rust, *Cronartium comptoniae* (anamorph: *Peridermium comptoniae*) attacks mainly *Pinus banksiana, P. contorta* and *P. resinosa* in North America (APS 1997).

Cronartium conigenum causes rust of most hard pine species (*P. maximinoi, P. pseudostrobus* and *P. oocarpa*) in Mexico and Central America (Sanchez-Ramirez *et al.* 1986; Rayachhetry *et al.* 1995).

Other hard pine stem rusts include: *Cronartium appalachianum*, attacking *Pinus virginiana* in eastern USA, and *Peridermium bethelii*, infecting *Pinus contorta* and *P. ponderosa* in western USA (APS 1997). Other species of rust fungi, such as *Cronartium himalayense*, which attacks *P roxburghii* in India (Shukla 1995) and *Cronartium* spp., infecting *P. massoniana* and *P. hwangshanensis* in China (Fang and Liu 1992), are also mentioned in the existing literature.

Pine needle rusts may be caused by numerous species of *Coleosporium* spp. Many of these species are morphologically indistinguishable. China contains half of the species of *Coleosporium* described (Xue-Yu 1996). Pine needle rusts are not considered major pathogens of pines (APS 1997). However, most *Pinus* species can be infected by *Coleosporium* spp., such as *P. sylvestris, P. heldreichii, P. caribaea, P. contorta, P. echinata, P. banksiana, P. thunbergii, P. elliottii, P. glabra, P. halepensis, P. kesiya, P. mugo, P. nigra, P. palustris, P. flexilis, P. pinaster, P. pinea, P. cembroides, P. roxburghii, P. yunnanensis, P. virginiana, P. wallichiana, P. brutia, P. palustris* and *P. resinosa* (Xue-Yu 1996; University of Sarajevo 1974; Kaneko *et al.* 1995; Hopkin and Howse 1994). Among the species of *Coleosporium* reported as pathogens of *Pinus* spp., *C. senecionis, C. asterum, C. apocynaceum, C. campanulae, C. crowellii, C. delicatulum, C. inulae, C. ipomoeae, C. pinicola, C. tussilaginis, C. vernonae* and *C. elephantopodis*, are mentioned (Butin and Kowalski 1989; Hopkin and Howse 1994; Kaneko *et al.* 1995). These species are summarized in Appendix 1.

Significance

This is one of the most important groups of pine diseases, resulting in the death of seedlings and trees and causing stem deformity. Fusiform rust destroyed a large proportion of pines (*Pinus taeda, P. elliottii, P. palustris*) in the southern USA (APS 1997). White pine blister rust

has been the most devastating disease in the white pine stands of the USA and Canada. Galls of the Western gall rust increase in size annually: thus infections on the main stem can kill seedlings or young trees. *Coleosporium* rusts may defoliate pines in nurseries and young plantations, causing unsightly foliage and slowing growth.

Symptoms and signs

Cronartium rusts: Symptoms on pines, the aecial host, include yellow-brown, diamond to elliptical-shaped cankers or swellings on trunks or branches (Fig. 24). These infections usually produce conspicuous amounts of resin. Galls, top and branch dieback (flagging), trunk bushiness, and breakage at the lesion and canker are also typical of several *Cronartium* diseases. The characteristic signs are white to orange-yellowish, blister-like aecia on the swollen or cankered organs (Figs 25–27). Yellow-orange masses of aeciospores also form on cankers. On the lower surface of alternate-host leaves, yellow to orange uredinial pustules develop (Figs 28 and 29), sometimes associated with chlorotic or necrotic areas. However, the most typical signs are the hair- or horn-like rose-cream to dark brown columnar telia, which can be seen alone (Fig. 30) or among the uredinial pustules (Fig. 31).

Endocronartium rusts: Woody, globose or pear-shaped perennial galls, reaching up to 30 cm in diameter, with orange-yellowish aeciospores (Fig. 32).

Fig. 24 (top). Fusiform rust galls on pine seedlings. (Dr R. Anderson, USDA Forest Service, Asheville)

Fig. 25 (left). Blister-like aecia of white pine blister rust *Cronartium ribicola* on the swollen branch of *Pinus strobus*. (Dr R. Anderson, USDA Forest Service, Asheville)

Fig. 26 (right). Aecia of eastern gall rust *Cronartium quercuum* f. sp. *echinatae* on *Pinus* sp. (Dr R. Anderson, USDA Forest Service, Asheville)

Fig. 27. Gall and aecia of fusiform rust *Cronartium quercuum* f. sp. *fusiforme* on *Pinus* sp. (Dr R. Anderson, USDA Forest Service, Asheville)

Coleosporium needle rusts: In spring, chlorotic to yellow spots or bands, which exude pycnidial drops, appear on pine needles. Later white, tongue-shaped or blister-columnar aecia form and produce orange-yellow aeciospores (Fig. 33). These disappear in late summer, leaving tiny scars or yellow or brown spots or bands on green or yellow needles. On the lower surface of alternate-host leaves, e.g. species of *Aster* and *Solidago*, yellow to orange uredinial pustules and orange to waxy dark-crusty or columnar cushion-like waxy telia appear. For some autoecious, microcyclic species, only columnar cushion-like telia form on needles (Ziller 1974; Cummins and Hiratsuka 1983).

Fig. 28 (left). Yellow-orange uredinial pustules of *Cronartium ribicola* on the alternate host *Ribes* sp. (Dr R. Anderson, USDA Forest Service, Asheville)

Fig. 29 (right). Yellow-orange uredinial pustules of *Cronartium quercuum* f. sp. *fusiforme* on the alternate host *Quercus* sp. (Dr R Anderson, USDA Forest Service, Asheville)

Biology and transmission

As an example, the life cycle of the heteroecious rust *Cronartium ribicola* is as follows: branch and stem infections are initiated by hyphal growth from the needles. One to several years later, in summer through autumn, spermogonia appear on the bark of affected tissues and in the following spring conspicuous aecia are produced on the bark of cankered, or swollen stems or branches, or both, where spermogonia occurred. Aeciospores infect alternate hosts (*Ribes* spp.), where uredinia are formed. On pines, after aeciospore dispersal, the blister aecia gradually dry and disappear, and the rust survives as hyphae in the bark surrounding the lesion or canker, where it sporulates annually. Urediniospores continue to be produced on alternate-host leaves and perpetuate and increase the rust on these hosts. In late summer through early autumn, urediniospore

Fig. 30 (top). Brown hair-like telia of *Cronartium quercuum* f. sp. fusiforme on the lower surface of *Quercus* sp. (Dr R. Anderson, USDA Forest Service, Asheville)

Fig. 31 (bottom). Telia of *Cronartium quercuum* f. sp. *fusiforme* among uredinial pustules on the lower surface of *Quercus* sp. (Dr R. Anderson, USDA Forest Service, Asheville) infections result in production of hair- or horn-like telial columns, alone or among the urediniospores. Basidiospores are liberated from the teliospore horns and dispersed by wind to pines, where they infect young needles.

The life cycle of autoecious rusts such as *Endocronartium harknessii* and some *Peridermium* spp. is completed on pine, i.e. no alternate host is involved. However, under some conditions, autoecious rusts seem to behave as facultative heteroecious (Gibbs *et al.* 1986; Moricca *et al.* 1996).

Detection
Look for the typical symptoms and signs described above.

Fig. 32 (left). Jack pine (*Pinus banksiana*) seedlings affected by western gall rust (*Endocronartium harknessii*), healthy seedling on right. (Dr J. Sutherland, Applied Forest Science, Victoria)

Fig. 33 (right). Aecia of needle rust *Coleosporium* sp. on pine needles. (Dr R. Anderson, USDA Forest Service, Asheville)

Nematode-caused disease

Pine wilt disease

Causal organism
Bursaphelenchus xylophilus (Steiner & Buhrer) Nickle.

Hosts
Many *Pinus* spp., including *P. echinata* (Linit and Kinn 1996); *P. taeda* (Dwinell *et al.* 1995); *P. densiflora* (Nakamura *et al.* 1995); *P. thunbergii* (Ikeda and Kiyohara 1995); *P. massoniana, P. strobus, P. palustris* (Suga *et al.* 1993); *P. sylvestris* (Sikora and Malek 1991) and other Pinaceae.

Geographical distribution
North America (Linit and Kinn 1996; Futai and Sutherland 1989), mainland China (Baojun and Qouli 1989; Zhu and Yao 1992) and Taiwan (Province of China), Japan (Ishida *et al.* 1993; Fujihara 1996), Korea (Choi and Moon 1989).

Significance
This disease kills pine trees in forests and landscape plantings. *Pinus* species vary in their susceptibility to the disease, with Japanese red pine (*Pinus densiflora*) and Japanese black pine (*P. thunbergii*) being particularly susceptible (Fig. 34). Pine wilt is the most destructive pine disease in Japan and China.

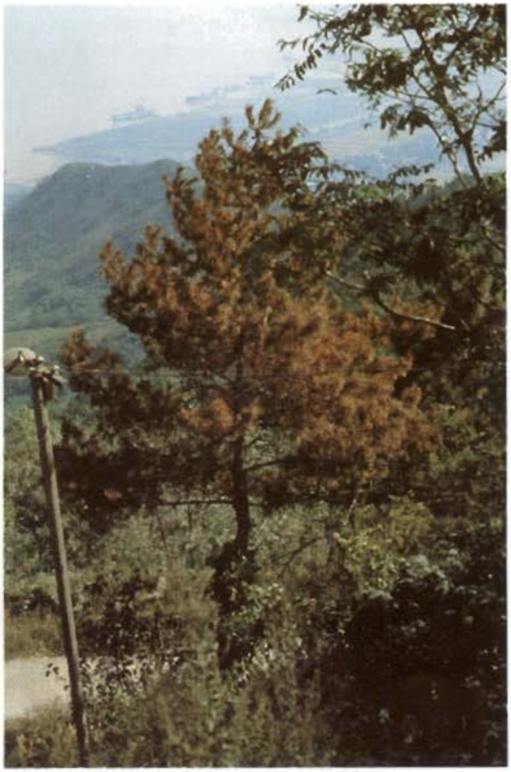

Fig. 34. Japanese black pine, *Pinus thunbergii* Parl., affected by pine wilt disease, Nanjing, People's Republic of China. (Dr J. Sutherland, Applied Forest Science, Victoria)

Symptoms and signs

Initial symptoms appear in summer through early autumn, and include yellowing and needle wilting. Usually trees die rapidly, but in cooler areas disease development may be slower and affected trees may survive until the following year. Dead trees characteristically exhibit reddish-brown foliage throughout the crown. No oleoresin flows from wounds made to the trunk, branches or twigs of diseased trees.

Biology and transmission

The nematode is vectored by wood-boring beetles of the family Cerambycidae, e.g. *Monochamus alternatus* in China, Korea and Japan, and *M. carolinensis, M. scutellatus* and *M. titillator* in the USA. These beetles become infested with the nematode just before emerging from diseased pine trees as adults. Individual beetles carry thousands of *B. xylophilus* dauerlarvae within their tracheae (Fig. 35). The beetles fly to healthy pines where they maturation-feed on the thin bark of twigs. There the nematodes leave the vector and enter the host tree via the feeding wounds. Dead trees are colonized by an array of fungi, particularly blue-stain fungi, upon which the nematodes feed and multiply. Female beetles then oviposit in dead trees where their larvae feed on the sapwood. Pine wilt disease is most prevalent in areas with warm temperatures, as the nematode completes its life cycle in 12, 6 and 3 days at 15, 20 and 30°C, respectively. Long-distance spread occurs with vector-infested logs. Since there may be a latency period between nematode infestation and symptom expression, germplasm materials such as scions could also contain the nematode.

Detection

Presence of wilt symptoms and tree mortality. The nematode can be easily obtained from pinewood by using Baermann funnels and propagated in the laboratory on cultures of fungi such as *Botrytis cinerea*.

Fig. 35. Fourth-stage larvae (dauerlarvae) of *Bursaphelenchus xylophilus* within and outside of trachea of *Monochamus alternatus*. (Dr. Y. Suto, Shimane Prefecture Forest Research Centre, Yatsuka-gun)

INSECTS AND SOME EXAMPLES RELEVANT FOR GERMPLASM MOVEMENT

Innumerable insects utilize pines as host material. Many pine feeding insects can be extremely damaging and many species have been accidentally introduced into areas where pines do not occur naturally and have become important forest plantation species (e.g. Australia, eastern and southern Africa, New Zealand and South America).

There are several records of accidental insect introductions with pine germplasm. The introduction of the European pine shoot moth, *Rhyacionia buoliana*, into the northeastern USA ca. 1941 was with pine seedlings imported from western Europe (Miller 1967). This insect has subsequently become a serious pest of young pine plantations throughout the northeastern and north central USA and adjoining portions of Canada. The introduction of this insect into western Oregon and Washington (USA) and British Columbia (Canada)

Fig. 36. *Pinus echinata* cone showing seeds damaged by seedworms (*Cydia toreuta*) and overwintering larval galleries. (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

during the 1960s is the result of within-country movement of nursery stock (Furniss and Carolin 1977). Introduction of the pine woolly adelgid, *Pineus boerneri*, into Kenya and Zimbabwe during the 1960s resulted from the introduction of infested scion material for tree improvement programmes (Varma 1996; Barnes *et al.* 1976). In 1988, a loblolly pine scale, *Oracella acuta*, was introduced with scion material into southern China from its natural range in the southeastern USA (Sun *et al.* 1996). This insect is a potential threat to extensive areas of exotic pine plantations in southeastern China.

Small insects pose the greatest risk of being moved to new locations with germplasm, especially members of the insect order Homoptera (e.g. aphids, mealybugs, and scales). As they feed on pine shoots, they may be introduced accidentally with germplasm in the form of scion material. Another group of pine insects worth mentioning are those attacking seeds and cones, e.g. *Dioryctria* spp. (Lepidoptera: Pyralidae), *Eucosma* spp. (Lepidoptera: Olethreutidae), *Conophthorus* spp. (Coleoptera: Scolytidae) and *Cydia* spp. (= *Laspeyresia* spp.) (Lepidoptera: Olethreutidae) (Fig. 36). This group of insects is well documented for North America (Hedlin *et al.* 1980) but is less well known elsewhere. Should seed and cone insects be introduced into new locations, they could potentially devastate pine seed production. Consequently, it is important to **never** ship cones containing seeds across international boundaries. Always extract seeds prior to shipment.

Examples of insects, which either have been documented to move, or have the potential to move, with pine germplasm, are described in the following sections.

Homoptera

Giant conifer aphids

Causal organisms
Cinara spp. (Homoptera: Lachnidae).

Hosts
Pinaceae and Cupressaceae. Some species confine their feeding to one genus of conifers, others feed on only a single species of host plant and others are general feeders. Many *Cinara* spp. infest various species of *Pinus*. *C. cronartii* restricts its feeding to lesions and cankers caused by the rust fungus *Cronartium quercuum* f.sp. fusiform on *P. taeda* and *P. elite* in the southeastern USA (Blackman and Eastop 1994). In South Africa, stems and roots of *P. taeda* and *P. patula* are infested (Kfir *et al.* 1985). *Cinara pinea* feeds on *P. sylvestris* (Kidd and Tozer 1985).

Geographical distribution
Of the 200 known species, 150 occur in North America, 30 in Europe and the Mediterranean Region and 20 in Asia. *Cinara* spp. have been introduced into Africa, Bermuda, New Zealand and South America (Blackman and Eastop 1994).

Significance
Several *Cinara* species that cause little or no damage in their native habitats have been introduced into new locations, where they are now causing serious damage. *C. cronartii* Tissot and Pepper has been introduced into South Africa (Kfir *et al.* 1985). *C. pinea* (Mordvilko) has been introduced into North America (Kidd 1988). Introduction of *C. juniperi* DeGeer into Bermuda resulted in damage to *Juniperus bermudiana*, an endemic species (Browne 1968). The introduction of *C. cupressivora* (initially identified as the synonym *C. cupressi* (Buckton) into eastern and southern Africa has resulted in devastating losses to plantations of *Cupressus lusitanica* (Ciesla 1991).

Damage
Yellow or reddish brown foliage, especially in the inner crowns of host trees; black sooty mould on twigs and foliage resulting from honeydew produced by aphid colonies; and tree mortality.

Biology
Infest roots, trunks, branches, twigs, shoots or foliage and usually produce several generations per year. There can be up to four adult forms: sexual winged, sexual wingless, parthenogenic winged and parthenogenic wingless. In temperate climates, sexual forms deposit eggs on foliage, shoots or bark. Eggs are the overwintering stage. They hatch into parthenogenic forms the following spring and produce live young until the onset of cooler temperatures, when a sexual form is produced. Species that have been

introduced into the tropics or subtropics lose their capacity to produce a sexual form and reproduce by parthenogenesis throughout the year. Most *Cinara* spp. feed in colonies of 20–80 adults and nymphs (Fig. 37). In areas of alternating wet and dry climates, *Cinara* colonies and tree damage tend to be more prevalent during dry periods. Could be moved with scion material or nursery stock. Once established in an area, dispersal occurs by air currents or winged adults.

Detection

Chlorotic or reddish-brown foliage, particularly in the inner crowns of host trees, and aphid colonies feeding on foliage or branches. Secondary indicators of infestation include black sooty mould, presence of larvae and adults of ladybird beetles (Family Coccinellidae), which are predators of *Cinara* spp., and ants that feed on honeydew and tend aphid colonies.

Fig. 37. Colony of giant conifer aphids (*Cinara* sp.) on *Pinus taeda* in South Carolina, USA. (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

Loblolly pine scale

Causal organism

Oracella acuta (Lobdell) (Homoptera: Pseudococcidae)

Hosts

Pinus echinata, P. palustris, P. virginiana and *P. taeda*, are hosts in the insect's natural range (Clarke *et al.* 1990). In China, *P. elliottii* is the main host, but *P. massoniana*, an indigenous species, is also infested (Ciesla 1994).

Geographical distribution

Southeastern USA (Clarke *et al.* 1990). Introduced into Guangdong Province, China (Sun *et al.* 1996).

Significance

Usually of minor importance in its native habitat but has recently appeared in large numbers in pine seed orchards following heavy use of chemical insecticides (Clarke *et al.* 1990). It was introduced into southeastern China in 1988 on pine scion material collected in the USA and grafted onto rootstocks in Guangdong Province. By June 1995, over 212 500 ha of pine plantations were infested (Sun *et al.* 1996).

Fig. 38. Shoot deformation due to infestation by the loblolly pine scale *Oracella acuta* on *Pinus elliottii* in Guangdong Province, China. (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

Damage
Heavy infestations result in premature abscission of foliage, reduced shoot growth and needle length and bud mortality (Fig. 38). Height growth may be reduced by 25–30%. Infestations can also occur on cones and cause deformity.

Biology
Pale rose nymphs and adults feed on buds and expanding shoots and produce white resin cells, which are used as protective cover (Fig. 39). Pale orange eggs are laid in clusters underneath resin cells. Honeydew from scales provides a medium for growth of black sooty mould on infested branches and foliage.

In its natural range, *O. acuta* has 4–5 generations per year, overwintering as crawlers under resin cells. There are at least that many generations in southern China. Sexual reproduction may occur but most reproduction is by parthenogenesis and most adults are females. Males are winged, females are wingless. Dispersal is by air currents moving immature crawlers from tree to tree. Can be moved on scion material. Once established, air currents easily spread insects.

Detection
Presence of resin cells on shoots and cones; black sooty mould; and reduced shoot growth.

Fig. 39. Loblolly pine scale (*Oracella acuta*) infestation on *Pinus elliottii* with white resin cells. (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

Pine bast scales

Causal organism
Matsucoccus spp. (Homoptera: Margarodidae)

Hosts
Pinus spp. (Appendix II).

Geographical distribution
Europe (Abgrall and Soutrenon 1991), China (McClure *et al.* 1983), Japan (Takenati 1972), North America (Drooz 1985), and Near East (Mendel 1988).

Significance
M. resinosae Bean and Godwin causes tree mortality in pine plantations in portions of the northeastern USA and may have been introduced (McClure *et al.* 1983).

Damage
The pest is capable of killing or weakening trees and making them susceptible to bark beetles (Coleoptera: Scolytidae). Infested trees may have dead tips, branch flagging, resinosis, stunting, as well as chlorotic and drying foliage.

Biology
Matsucoccus spp. nymphs, larvae and adults are small, oval, yellow to brown, inconspicuous insects that are difficult to detect because they push themselves beneath the sheath of needle fascicles or bury themselves in crevices of the bark of twigs and branches. Adult males are winged, females lack wings. They often take on the colour of their surroundings. Some species produce white waxy secretions and have black sooty moulds associated with them.

Life cycles of many species are similar. *M. feytaudi* has one generation per year. Adult females lay eggs in late winter–early spring. Motile nymphs occur from April to May and sessile nymphs are present until September, when they transform into pre-adults, the overwintering stage. Adults are present in late winter–early spring. *M. resinosae* has two generations per year in the northeastern USA.

They could be transported with scion material. Once established, they could be spread in air currents.

Detection
Presence of dead tips or branches, scale insects on surface of needles, shoots or bark.

Pine needle aphid

Causal organism
Eulachnus rileyi (Williams) (Homoptera: Lachnidae)

Hosts
Pinus spp. In Europe, *P. montana* and *P. nigra* are preferred over *P. sylvestris* (Blackman and Eastop 1994). In eastern and southern Africa, *P. caribaea*, *P. chiapensis*, *P. elliottii*, *P. kesiya*, *P. merkusii*, *P. michoacana*, *P. oocarpa*, *P. patula*, *P. roxburghii* and *P. taeda* are hosts (Murphy *et al*. 1991).

Geographical distribution
Europe and Asia; introduced into North America and eastern and southern Africa (Blackman and Eastop 1994).

Significance
This insect has caused only minor damage where it has been introduced, e.g. in North America and eastern and southern Africa. However, it has the potential to cause serious damage.

Fig. 40. Pine needle aphid *Eulachnus rileyi*: adult and late stage nymph. (Dr A. Cross, CABI-Bioscience, Ascot)

Damage
Heavy infestations cause needles to turn yellow and drop prematurely, resulting in growth reduction.

Biology and spread
Adults are spindle-shaped, 2.5 mm in length and dark olive green to orange brown or grey and are covered with a dusting of bluish-grey wax (Fig. 40). They produce copious quantities of honeydew. All life stages feed on the underside of pine needles. In temperate climates, both sexual and asexual forms exist. Adults are normally wingless, but winged forms are sometimes produced. In Africa, the species has a reduced life cycle, with only asexual forms occurring. Populations tend to increase during dry periods (Murphy *et al.* 1991). These insects could be moved with scion material. Once established in a new location, they are subject to wind dispersal.

Detection
Wax-covered aphids on underside of pine needles, along with honeydew and sooty mould.

Pine tortoise scale

Causal organism
Toumeyella parvicornis (Cockerell) (= *T. numismatica* Pettit & McDaniel) (Homoptera: Coccidae).

Hosts
Pinus elliottii, P. echinata and *P. taeda* (Fatzinger *et al.* 1992; Clarke *et al.* 1992).

Geographical distribution
Eastern North America (Drooz 1985).

Significance
Feeds on foliage. Heavy infestations damage seedlings, saplings and occasionally mature trees. Repeated infestations can kill trees. There has been no evidence of movement of this insect to date, but it has potential for transfer with germplasm.

Damage
Infested trees may have chlorotic or abnormally short needles and dead branches. In some cases, entire trees may be killed. Infestations may be accompanied by honeydew and black sooty mould.

Biology and spread
Adults are reddish-brown, oval, convex soft scales, 5–7 mm long on the stems of host trees. There is one generation per year in the northern part of its range. Two generations have been documented for Maryland, USA, and there may be additional generations per year further south. In the north, this insect overwinters as fertilized females on the stems of host trees. By late spring, females enlarge and about 500 eggs are laid in early summer. Eggs hatch shortly after oviposition and first instar nymphs develop into adults in mid- to late summer. Can be transmitted with scion material. Once infestations are established, crawlers are easily dispersed by wind.

Detection
Presence of insects; chlorotic or abnormally short needles; and dead branches.

Pine woolly adelgids

Causal organism
Pineus spp. (Homoptera: Adelgidae).

Hosts
Includes *Pinus halepensis*, *P. brutia* (Mendel *et al.* 1994), *P. caribaea*, *P. oocarpa* (Mailu *et al.* 1982), *P. sylvestris* (Heliovaara and Vaisanen 1989), *P. resinosa* (McClure 1989), *P. ponderosa* (Latta and Linhart 1997), *P. radiata* (Blackmann *et al.* 1995), *P. strobus* (Montgomery *et al.* 1996, *P. pinaster* (Zwolinski *et al.* 1989) and *P. patula* (Madoffe and Austara 1993), *P. tabulaeformis*, *P. thunbergii* (McClure 1984) and *Picea* spp. Primary hosts of sexual forms of *Pineus* spp. are *Picea*, on which terminal compact galls, which superficially resemble cones, are formed. *Pinus* spp. are considered a secondary host of the asexual forms of most *Pineus* spp.

Fig. 41. Shoot of *Pinus patula*, infested by the pine woolly adelgid *Pineus boerneri*, near Kabale, Uganda. (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

Geographical distribution

The 21 recognized species are widely distributed throughout conifer forests in the Northern Hemisphere. *Pineus boerneri* was accidentally introduced into Australia, Africa, New Zealand, South America and USA (Hawaii and northeastern states). *P. pini* has been introduced into North America, Australia and New Zealand and *P. strobi* has been introduced into Europe (Blackman and Eastop 1994).

Significance

Some *Pineus* spp. have been accidentally introduced into areas where pine trees have been introduced as plantation trees, and caused severe damage. Accidental introduction of *P. boerneri* Annand (initially misidentified as *Pineus pini* MacQuart) into Kenya and Zimbabwe probably took place on scion material (Odera 1974, Barnes *et al.* 1976).

Damage

Nymphs and adults suck plant juices from needles, shoots or stems of pine and cause shoot deformity and loss of height growth. Excess plant juice excreted by adelgids as honeydew is a favourable medium for growth of black sooty moulds on foliage, shoots and stems.

Biology

Some species are holocyclic, alternating their hosts between *Picea* and *Pinus*, to complete a cycle with seven life stages over two years. Sexual forms on *Picea* produce galls on branches. Asexual forms on either *Pinus* or *Picea* occur on foliage, shoots or bark (exception: *P. abietinus* on *Abies* spp.). For other species, e.g. *P. boerneri, P. strobi,* only the asexual form is known. Adults of pine-infesting forms are covered with a conspicuous white, flocculent wool (Fig. 41). They can be moved with scion material. Once established, air currents easily spread them.

Detection

Presence of foliage or stems covered with white, waxy masses; deformed shoots; and black sooty mould.

Lepidoptera

Pine shoot moths

Causal organism
Rhyacionia spp. (Lepidoptera: Tortricidae).

Hosts
Pinus spp. (summarized in Appendix III).

Geographical distribution
Europe, Japan, North and Central America (Appendix III). *R. buoliana* has been introduced into North America and Argentina, Chile and Uruguay in South America (Abgrall and Soutrenon 1991; Browne 1968; Drooz 1985; Kobayashi 1962; Cibriàn Tovar 1995; Furniss and Carolin 1977).

Fig. 42. *Pinus taeda* showing shoot damage by *Rhyacionia frustrana* (Nantucket pine tip moth). (Dr W.M. Ciesla, Forest Health Management International, Fort Collins)

Significance

The European pine shoot moth, *R. buoliana* Denis and Schiffermüller, is a major pest of saplings, young trees and short-rotation pine plantings (e.g. pulpwood and Christmas trees). This insect has been introduced into North and South America, where it has become very damaging. It recently appeared in Chile and is spreading rapidly through that country's extensive industrial *Pinus radiata* plantations (Beeche Cisternas *et al.* 1992). Nantucket pine tip moth, *R. frustrana* (Comstock), is a pest of young pine plantations in the eastern USA. Subtropical pine tip moth, *R. subtropica* Miller, has caused serious losses of grafted *P. elliottii* scions in tree improvement programmes in the Southeastern USA.

Damage

Buds and shoots of pine trees are infested, causing deformity and reduced height growth. Infested shoots have reddish-brown needles (Fig. 42) and dead, frass-filled buds, often containing larvae and pupae.

Biology

Adults have an average wingspan of about 15–20 mm. Forewings are marked with rusty, orange-red or brick red patches. Head, body and appendages are covered with grey scales (Fig. 43). Mature larvae are 9–12 mm long and range in colour from dark red-brown to yellowish with black heads (Fig. 44). Reddish-brown pupae typically protrude from damaged shoots prior to adult emergence.

Fig. 43. European pine shoot moth (*Rhyacionia buoliana*) adult. (Dr D. LanFranco, Universidad Austral de Chile, Valdivia).

R. buoliana has one generation per year, with moths flying in mid summer. Eggs are laid singly or in pairs on buds, needle sheaths or shoots of hosts. Young larvae mine in buds, making silk lined tunnels. Surface of buds may also be covered with silk. Larvae overwinter in buds and shoots. They become voracious feeders the following spring, moving from bud to bud. Pupation occurs in mined tissues (Browne 1968). Some North American species (e.g. *R. frustrana*, and *R. bushnelli*) have at least 2–3 generations per year and overwinter as larvae or pupae in the pine debris. Introduction of *R. buoliana* into North America ca 1914 is believed to have occurred on nursery stock imported from western Europe (Miller 1967).

Detection

Occurrence of dead shoots and buds filled with frass, larvae or pupae.

Fig. 44. European pine shoot moth (*Rhyacionia buoliana*) larva in *Pinus radiata* shoot. (Dr D. LanFranco, Universidad Austral de Chile, Valdivia).

Coleoptera

Pine shoot beetle

Causal organism
Tomicus piniperda (L.) (= *Blastophagus piniperda* (L.) (Coleoptera: Scolytidae).

Hosts
Primarily a pest of *P. sylvestris* in Europe (Mazur and Perlinski 1992; Langstrom and Hellquist 1993), northern Asia (Kolomiets and Bogdanova 1992), and N. America (Czocajlo *et al.* 1997).

Geographical distribution
Conifer forests of Europe and northern Asia (Kolomiets and Bogdanova 1992). Recently introduced into North America and now established over a large area of the north central USA and adjoining Canada (Haack *et al.* 1993).

Significance
A major bark beetle pest in Europe, and it has subsequently been introduced into North America (Haack and Lawrence 1995).

Damage
Adults feed in shoots of pine trees and other conifers prior to overwintering, mating and reproduction.

Biology and spread

Adults are small, cylindrical, dark brown to black beetles, 3.5–5 mm long, with clubbed antennae. There is one generation per year; adults fly in early spring. Breeding occurs in the cambium of weakened and/or dying conifers, stumps, logs or down trees. Emerging adults feed in tips and shoots of mature pine trees and other conifers prior to mating and reproduction (maturation feeding). Overwintering occurs in thick bark at base of pine trees. Adults feeding in shoots could be moved with scion material. Adults are strong fliers and spread rapidly once established in a new area.

Detection

Shoots with discoloured foliage, resin and boring dust; adult beetles in shoots (Fig. 45).

Fig. 45. Pine shoot infested by *Tomicus piniperda*: resin and adult beetles in shoot. (Dr B. Langstrom, Swedish University of Agricultural Sciences, Garpenberg)

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APPENDIX I. HOSTS AND GEOGRAPHICAL DISTRIBUTION OF PINE RUSTS (*CRONARTIUM* AND *COLEOSPORIUM* GROUPS)

Pathogen and common name	Hosts	Alternate hosts and geographical distribution
Cronartium coleosporioides Arth. syn. *C. stalactiforme*, *C. filamentosum*, *Peridermium stalactiforme* stalactiform rust, cow wheat rust	*P. attenuata*, *P. banksiana*, *P. contorta*, *P. echinata*, *P. mugo*, *P. ponderosa*, *P. sylvestris*, *P. halepensis*, *P. coulteri*, *P. jeffreyi*	*Castilleja*, *Melampyrum*, *Orthocarpus*, *Pedicularis*, *Rhinanthus* spp. Canada, USA
Cronartium comandrae Peck. syn. *C. asclepiadeum* var. *thesi*, *C. pyriforme*, *Peridermium pyriforme* and *P. thesi* comandra rust	*P. attenuata*, *P. banksiana*, *P. contorta*, *P. echinata*, *P. eldarica*, *P. elliottii var. elliottii*, *P. glabra*, *P. jeffreyi*, *P. mugo*, *P. ponderosa*, *P. rigida*, *P. serotina*, *P. sylvestris*, *P. taeda*, *P. virginiana*	*Comandra* spp., mainly *C. pallida*, *C. umbellata*, *C. richardsoniana* and *Geocaulon lividum* (= *C. livida*) Canada, USA
Cronartium comptoniae Arth. sweet-fern blister rust	*P. banksiana*, *P. contorta*, *P. coulteri*, *P. densiflora*, *P. echinata*, *P. jeffreyi*, *P. muricata*, *P. mugo*, *P. nigra*, *P. pinaster*, *P. ponderosa*, *P. pungens*, *P. radiata*, *P. resinosa*, *P. rigida*, *P. sylvestris*, *P. taeda*, *P. virginiana*	*Comptonia peregrina*, *Myrica gale*, *M. cerifera*, *M. carolinensis* Canada, USA
Cronartium flaccidum (Alb. & Schw.) Winter syn. *C. asclepiadeum*, *C. euphrasiae*, *Peridermium pini* Scots pine blister rust	*P. densiflora*, *P. halepensis*, *P. montana*, *P. pinea*, *P. pinaster*, *P. sylvestris*, *P. wallichiana*, *P. kesiya*, and many others	*Asclepias*, *Cynanchum*, *Euphrasia*, *Impatiens*, *Gentiana*, *Loasa*, *Melampyrum*, *Nemesia*, *Paeonia*, *Pedicularis*, *Ruellia*, *Schizanthus*, *Tropaeolum*, *Verbena Vincetoxicopsis* spp. Europe and Asia

(cont.)

Pathogen and common name	Hosts	Alternate hosts and geographical distribution
Cronartium himalayense B.K. Bakshi syn. *Peridermium himalayense* chir pine blister rust	*P. roxburghii* and *P. canariensis*	*Swertia* spp. India, Pakistan, Philippines
Cronartium quercuum (Berk.) Miyabe ex Shirai syn. *C. cerebrum, Peridermium giganteum, P. mexicanum* pine-oak gall rust, eastern gall rust	*P. banksiana, P. chihuahuana, P. clausa, P. densiflora, P. echinata, P. elliottii, P. kesiya, P. luchuensis, P. massoniana, P. montezumae, P. mugo, P. nigra, P. oocarpa, P. patula, P. pinaster, P. ponderosa, P. resinosa, P. rigida, P. serotina, P. sylvestris, P. tabulaeformis var. mukdensis, P. taeda, P. thunbergii and P. virginiana*	*Quercus* spp. *Castanea crenata, Castanopsis cuspidata,* Europe, N. America, Japan *Castanea* spp. *Pasania* spp. North America India
Cronartium quercuum Miyabe ex Shirai f. sp. *fusiforme* syn. *C. fusiforme, Peridermium fusiforme* fusiform rust	*P. caribaea, P. contorta, P. elliottii* var. *densa, P. jeffreyi, P. nigra, P. palustris, P. ponderosa, P. pseudostrobus, P. radiata, P. rigida, P. serotina, P. sylvestris, P. taeda*	*Quercus* spp. USA
Cronartium ribicola syn. *C. ribis, Peridermium strobi* white pine blister rust	*P. albicaulis, P. armandii, P. ayacahuite, P. cembra, P. flexilis, P. koraiensis, P. lambertiana, P. monticola, P. pumila, P. strobus, P. wallichiana*	*Grossularia* spp., *Ribes* spp., *Pedicularis* spp. Canada, China, India, Iran, Japan, Korea, Russia, Taiwan, USA
Endocronartium harknessii (J. P. Moore) Y. Hiratsuka syn. *Cronartium harknessii, Peridermium cerebroides, P. harknessii* western gall rust	*Pinus attenuata, P. banksiana, P. canariensis, P. caribaea, P. contorta var. latifolia, P. elliottii, P. engelmannii, P. halepensis, P. jeffreyi, P. mugo, P. muricata, P. nigra, P. pinaster, P. ponderosa, P. radiata, P. sabiniana, P. sylvestris*	autoecious North America

(cont.)

Pathogen and common name	Hosts	Alternate hosts and geographical distribution
Endocronartium pini (Pers.) Y. Hiratsuka, syn. *Peridermium pini*	*P. halepensis, P. mugo, P. nigra, P. pinaster, P. sylvestris*	autoecious Europe
Endocronartium sahoanum Imazu and Kakish.	*P. pumila*	autoecious Japan
Endocronartium yamabense (Saho and Takahashi) Paclt	*P. pumila*	autoecious Japan
Coleosporium apocynaceum Cooke	*P. taeda, P. elliottii, P. palustris*	*Amsonia* spp. Korea, Russia, Taiwan, USA
Coleosporium asterum (Dietel) Syd. & P. Syd. red pine needle cast	*P. banksiana, P. contorta, P. coulteri, P. densiflora, P. massoniana, P. ponderosa, P. pungens, P. resinosa, P. sylvestris, P. taeda, P. thunbergii* and *P. yunnanensis*	*Aster* spp., *Solidago* spp. Bermuda, China, Europe, Korea, Japan, N. America, Russia, Taiwan
Coleosporium barclayense B.K. Bakshi Himalayan pine needle rust	*P. griffithii* and *P. wallichiana*	*Senecio* spp. India, Pakistan
Coleosporium campanulae Leb. ex Kickx. fil.	*P. banksiana, P. densiflora, P. griffithii, P. nigra, P. resinosa, P. rigida, P. roxburghii, P. sylvestris, P. thunbergii*	*Campanula* spp., *Lysimachia* spp., *Specularia* spp. Asia, Europe, North America
Coleosporium crowellii Cummins syn. *Gallowaya crowellii*	*P. ayacahuite, P. cembroides, P. flexilis, P. montezumae*	autoecious Mexico, USA
Coleosporium delicatulum Arth.	*P. echinata, P. elliottii, P. nigra, P. palustris, P. resinosa, P. rigida, P. serotina, P. taeda*	*Euthamia* spp. USA

(cont.)

Pathogen and common name	Hosts	Alternate hosts and geographical distribution
Coleosporium inulae (Kunze) E. Fisch.	*P. halepensis, P. pinaster, P. pinea, P. roxburghii* and *P. sylvestris*	*Inula* spp. Europe, Palestine, Canary Islands, North Africa (in the Congo and India only on the alternate hosts)
Coleosporium ipomoeae (Schwein.) Burrill	*P. edulis, P. palustris, P. echinata, P. elliottii, P. leiophylla, P. rigida, P. serotina, P. taeda*	*Convolvulus* spp., *Ipomoea* spp. USA (in Central and South America only on the alternate hosts)
Coleosporium pinicola Arth. syn. *Gallowaya pinicola, G. pini*	*P. banksiana, P. brutia, P. halepensis, P. nigra, P. pinea, P. sibirica, P. virginiana*	autoecious Canada, Cyprus, Russia, USA
Coleosporium tussilaginis (Pers.) Lev. According to Gibson (1979) a complex of heteroecious rusts including *C. cacaliae* (DC.) Furchal, *C. campanulae* Lev., *C. euphasiae* (Schum.) Wint., *C. melampyri* Tul., *C. narcissi* Grove, *C. petasitis* Lev., *C. rhinantheracearum* Lev., *C. senecionis* Fr. ex. Nick., *C. sonchi* (Straus) Lev. and *C. tropaeoli* Desm. pine needle rust	2- and 3-needle pines	*Cacalia* spp., *Campanula* spp., *Clerodendron* spp., *Euphrasia* spp., *Melampyrum* spp., *Narcissus* spp., *Petasites* spp., *Rhinanthus* spp., *Senecio* spp., *Sonchus* spp., *Tropaeolum* spp. and *Tussilago* Europe, India and Philippines?, Canada? and South America (in Argentina and Brazil only on the alternate hosts)
Coleosporium vernonae Berk & M. A. Curtis.; syn. *C. paraguayense, C. elephantopodis, Uredo elephantopodis*	*P. caribaea, P. contorta, P. echinata, P. elliottii, P. glabra, P. halepensis, P. kesiya, P. mugo, P. nigra, P. palustris, P. pinaster, P. pinea, P. rigida, P. roxburghii, P. taeda, P. yunnanensis, P. virginiana* and *P. wallichiana*	*Elephantopus* spp. *Vernonia* spp. China, West Indies, USA; (In Argentina and Brazil only on the alternate hosts)

APPENDIX II. HOSTS AND GEOGRAPHICAL DISTRIBUTION OF IMPORTANT *MATSUCOCCUS* SPECIES

Species	Hosts	Geographical distribution	Reference(s)
M. acalyptus Herbert	*P. aristata, P. balfouriana, P. edulis, P. lambertiana*	Southwestern USA	Christensen *et al.* 1977
M. feytaudi Duc.	*P. pinaster*	France, Spain	Abgrall and Soutrenon 1991
M. josephi Bodenheimer & Haipaz	*P. halepensis*	Israel	Mendel 1988
M. matsumurae (Kuwana)	*P. densiflora, P. massoniana, P. tabulaeformis, P. taiwanensis, P. thunbergii*	China‡, Japan	McClure *et al.* 1983; Takenati 1972
M. resinosae Bean and Godwin†	*P. densiflora, P. resinosa*	Northeastern USA§	McLure 1990
Matsucoccus sp. nov.	—	Korean Peninsula	Miller & Park 1987
M. vexillorum Morrison	*P. thunbergii, P. ponderosa*	Southwestern USA	Christensen *et al.* 1977

†May be synonymous with *M. matsumurae*. ‡Introduced. §May have been introduced.

APPENDIX III.
HOSTS AND GEOGRAPHICAL DISTRIBUTION OF IMPORTANT SPECIES OF *RHYACIONIA*

Species	Hosts	Geographical distribution	Reference(s)
R. buoliana Dennis & Schiffermüller	*P. banksiana, P. brutia, P. contorta, P. elliottii, P. montana, P. nigra, P. pinaster, P. radiata, P. resinosa, P. rigida, P. sylvestris, P. taeda*	Europe, North† and South America†	Abgrall and Soutrenon 1991; Browne 1968; Drooz 1985
R. bushnelli (Bush)	*P. banksiana, P. ponderosa, P. resinosa, P. sylvestris*	Central North America	Drooz 1985
R. duplana simulata Heinrich	*P. caribaea, P. densiflora, P. elliottii, P. rigida, P. thunbergii*	Japan	Kobayashi 1962
R. frustrana (Comstock)	All pines within insect's natural range.	Eastern USA	Drooz 1985
R. neomexicana (Dyar)	*P. arizonica, P. ponderosa*	Southwestern USA, Mexico	Cibriàn Tovar 1995; Furniss and Carolin 1977
R. subtropica Miller	*P. caribaea, P. elliottii, P. palustris, P. taeda*	Southeastern USA	Drooz 1985

†Indicates accidental introduction and establishment.

APPENDIX IV.
COMMENTS ON TECHNICAL GUIDELINES FOR THE SAFE MOVEMENT OF *PINUS* GERMPLASM

please send to:

Germplasm Health Scientist
IPGRI-Americas
AA 6713, Cali, Colombia
Fax: 57-2-4450073

or Forest Resources Development Service
FAO
Via delle Terme di Caracalla
00100 Rome, Italy
Fax: +39-06-57055137

I would like to bring the following
[ ] inaccuracy(ies)
[ ] new development(s)
[ ] omission(s)
[ ] concerns
to the attention of the editors:
Disease ______________________________
Comments ______________________________

From:

Name

Address

Date

Signature

FAO/IPGRI Technical Guidelines for the Safe Movement of Germplasm are published under the joint auspices of the Plant Production and Protection Division of the Food and Agriculture Organization of the United Nations (FAO) and the International Plant Genetic Resources Institute (IPGRI).

The FAO Netherlands Partnership Programme provided funds for printing this document.

The International Plant Genetic Resources Institute (IPGRI) is an autonomous international scientific organization, supported by the Consultative Group on International Agricultural Research (CGIAR). IPGRI's mandate is to advance the conservation and use of genetic diversity for the well-being of present and future generations. The Institute operates through three programmes: (1) the Plant Genetic Resources Programme, (2) the CGIAR Genetic Resources Support Programme and (3) the International Network for the Improvement of Banana and Plantain (INIBAP). Financial support for the Research Agenda of IPGRI is provided by the Governments of Albania, Armenia, Australia, Austria, Belgium, Brazil, Bulgaria, Canada, China, Croatia, Cyprus, Czech Republic, Denmark, Estonia, Finland, France, Germany, Greece, Hungary, Iceland, India, Ireland, Israel, Italy, Japan, Republic of Korea, Lithuania, Luxembourg, Macedonia (F.Y.R.), Malta, the Netherlands, Norway, Peru, the Philippines, Poland, Portugal, Romania, Slovakia, Slovenia, South Africa, Spain, Sweden, Switzerland, Thailand, Turkey, Uganda, UK, USA and F.R. Yugoslavia (Serbia and Montenegro).

Citation: Diekmann, M., J.R. Sutherland, D.C. Nowell, F.J. Morales and G. Allard, editors. 2002. FAO/IPGRI Technical Guidelines for the Safe Movement of Germplasm. No. 21. *Pinus* spp. Food and Agriculture Organization of the United Nations, Rome/International Plant Genetic Resources Institute, Rome.

ISBN 92-9043-525-9