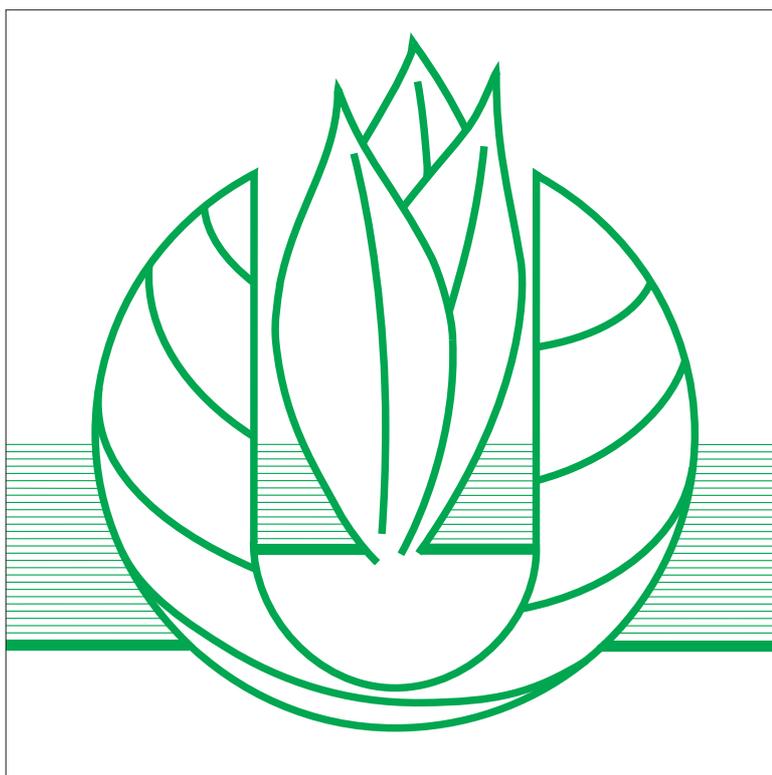# *Acacia* spp.

K.M. Old, T.K. Vercoe, R.B. Floyd, M.J. Wingfield, J. Roux and S. Neser

FAO

IPGRI

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
Sweet Potato	1989
Yam	1989
Legumes	1990
Cassava	1991
Citrus	1991
Grapevine	1991
Vanilla	1991
Coconut	1993
Sugarcane	1993
Small fruits (*Fragaria, Ribes, Rubus, Vaccinium*)	1994
Small Grain Temperate Cereals	1995
Musa spp. (2nd edition)	1996
Stone Fruits	1996
Eucalyptus spp.	1996
Allium spp.	1997
Potato	1998
Pinus spp.	2002

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INTRODUCTION

The collection, conservation and utilization of plant genetic resources and their global distribution and exchange 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 cause no symptoms 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 plant germplasm. In addition, current national regulatory information governing the export and importation of plant germplasm in the respective countries is essential.

The International Plant Protection Convention (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. As the depository of the IPPC, FAO collaborates with IPGRI to ensure and facilitate the safe movement of plant germplasm. The objective of the collaborative activities developed by FAO and IPGRI 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 major outputs of this collaborative programme include 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.

These technical guidelines are produced by panels of experts on the crop or plant concerned, selected in consultation with national and international research agricultural or forestry institutions working on the relevant crop group or plant species or genus.

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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'.

The experts contribute to the elaboration of the technical guidelines in their personal capacity and do not represent 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 participating specialists at the time of publication but the information provided needs to be regularly updated. The experts who contribute to the production of the technical guidelines are listed in this publication. Correspondence regarding this publication should be addressed to either FAO or IPGRI.

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 information given on a particular insect or disease 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 in germplasm 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 but these should be considered in addition to national and international phytosanitary measures.

The present guidelines were conceptualized first through the work of an interactive acacias mailing list, followed by a one day Preparatory Meeting for the Development of Technical Guidelines for the Safe Movement of Acacia Germplasm, hosted by the Forest Research Institute Malaysia (FRIM), on 20 March 1999; the participants list is provided thereafter. The guidelines were compiled by a group of specialists contracted by FAO and IPGRI (see List of Authors and Affiliations).

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.

PARTICIPANTS OF THE DEVELOPMENT OF TECHNICAL GUIDELINES FOR THE SAFE MOVEMENT OF ACACIA GERMPLASM, PREPARATORY MEETING, FRIM MALAYSIA, 20 MARCH 1999

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IMPORTANCE OF ACACIAS

Acacias (*Acacia* Mill.) are woody legumes of the Mimosaceae family, occurring naturally in all the inhabited continents except Europe. There are more than 1500 species, worldwide, with around 1200 of these endemic to Australia. Many are pioneer species in their natural environments and consequently, they usually display very vigorous growth in cultivation ranging in size from small shrubs to large forest trees. Acacias are suitable for a wide range of wood and non-wood uses. Like other legumes, they form symbiotic associations with soil bacteria to fix atmospheric nitrogen, making them suitable for poor quality sites. Information on growth, form and products are given in Boland 1989; Maslin *et al.* 1998; Turnbull 1986, 1987, and 1991; Doran and Turnbull 1997; Midgley and Turnbull 2001. There are several related taxonomic groups in the genus that may eventually be described as new genera. Each of these groups occurs primarily within one geographic region but, in some cases, there are overlaps into other regions. For the purposes of germplasm movement, the groups have similar characteristics but weed risk assessments would need to treat the groups differently. Also, the species most in demand for industrial plantations are of Australian origin.

Acacias are found in both extremely wet and in very dry ecosystems, and occur as either vigorous pioneers or as longer term, more established components of the vegetation. Most acacias have highly durable seeds capable of withstanding fire and remain viable during medium to long-term storage in the soil, under natural or planted stands. Some species have developed animal-attracting seed appendages that, together with thick seed coats, allow seed to be transported large distances in animal digestive tracts to aid dispersal. Some species also display root suckering and coppicing as responses, primarily, to defoliation by grazing and fire damage.

Acacias have been planted in more than 80 countries around the world but they have become particularly prominent in tropical and sub-tropical regions of Asia, Africa, Central and South America. There are more than 2 million hectares of commercial plantations in these regions as well as amenity and land-care plantings. Their suitability for marginal land and their vigorous pioneering growth habit coupled with prolific seed production has resulted in their ready uptake in industrial plantations. These characteristics have sometimes had negative consequences, with several species outstaying their welcome as intractable weeds. This potential for weediness should always be included in risk analysis prior to large scale planting.

Much of the existing plantation resource is based on seed from wild stands. Increasingly these plantations are being replaced by seed orchard-improved seed and in some cases clonal plantations from cuttings. The vigorous and precocious nature of the species that are used in industrial plantings has enabled rapid progress to be made in tree improvement programmes. For example in *A. crassicarpa*, second generation seed

orchard seed is now available within two decades of initial plantation trials and within a single decade of broad scale planting. Hybrids between *Acacia* species are also increasing as sources of plantation germplasm.

Germplasm movement

The large industrial acacia plantations of the world are based on acacias of Australian origin, although subsequent movement has occurred between many countries, independently of any Australian involvement. As the annually planted areas have rapidly increased there has been continuous demand for seed supply, leading to the transfer of large amounts of seed (tonnes) between countries, particularly in South-East Asia. The seeds of a number of species are edible (Maslin *et al.* 1998), which is resulting in additional transfer of seed between countries, particularly in sub-Saharan Africa, along with the native African species. Countries in South Asia (India in particular) and South and Central America, having assessed earlier trial plantings with promising results, are starting to request larger quantities of seed for commercial plantations. The CSIRO Australian Tree Seed Centre (ATSC) dispatched 1200 acacia seed-lots to 31 countries during 2000 representing 37 different species. A summary of transfers over the last 5 years is provided in Table 1.

Table 1. Records of *Acacia* seed-lots dispatched from the CSIRO Australian Tree Seed Centre during 1995–2000.

Recipient region	Number of species	Number of seed-lots
Europe	25	85
North Africa	41	209
Central Africa	22	122
Southern Africa	73	330
South Asia	27	544
Southeast Asia	53	4598
North Asia	48	1238
North America	16	93
Central America	16	94
South America	26	346
Pacific	14	100
Total	361	7759

Most transfers of genetic material are currently by seed, however, with the development of hybrids and selected clones, there are some movements of cuttings and tissue-cultured plants. These occur on a small scale as the initial transfer is commonly a few mother plants that are multiplied in the country of use. Increasing globalization of large-scale plantation forestry and processing companies is also having an impact on germplasm movement, with companies transferring elite material across international boundaries as part of their company operations.

The seeds of most species are highly resilient and present few problems for collection, storage and transport. The hard seed coats of many species allow harsher handling and quarantine treatment than many other tree species. If the seed is fully ripe at the time of collection, it is possible to remove extraneous material during processing. Thorough cleaning can be difficult if the seed is collected prior to full legume maturation. This does not necessarily affect seed viability in the short term, but it can adversely affect storage life and reduce pretreatments required to break dormancy. The funicles, arils and remaining legume fragments also provide refuges for insects and pathogens.

Acacia seed coats are usually hard and impervious to water and require scarification or heat treatment to promote germination. Experience at the ATSC has shown that pretreatment of seed with boiling water (for between 1 and 5 minutes) results in very little fungal contamination of the vermiculite substrate in germination dishes.

Conclusions

With the relatively rapid increase in acacia plantation area and the suitability of many species for very poor quality sites, the demand for seed is unlikely to decrease in the near future. Most of the currently utilized plantation species set large quantities of seed at a very early age, so it is possible for countries to produce enough seed to meet their requirements, within their own borders, fairly quickly. However, for the first 1–5 years, new plantation programmes will require seed from either wild populations or from existing plantations and seed orchards. Breeding programmes are starting to exchange elite clones by either cuttings or as tissue-cultured plants but so far this represents relatively small numbers.

The seed of most *Acacia* species will withstand relatively severe surface treatments without affecting short-term viability, which should assist the control of insect pests and pathogens in transit. Seed can be cleaned to a high purity eliminating the funicles and legume trash that can house pathogens.

Risk assessment of the movement of acacia seed should include evaluation of the potential for weed outbreak as well as the potential for pest and pathogen introduction.

GENERAL RECOMMENDATIONS

In common with other tree genera, the reproductive materials that are moved internationally include seed, cuttings, tissue-cultured plantlets, and perhaps in future, pollen. Each of these will be dealt with separately to provide the basis on which recommendations can be made.

Seed

Unlike some tree and crop species, where the term 'seed' may be used to describe reproductive propagules which are more properly regarded as fruits, acacias produce true seeds within legume pods. Seed can be liberated explosively during the drying of the pod or remain attached to funicles. Funicles and, for many tropical acacias, arils which develop after fertilization, may be retained by the seed after being shed. Arils may be highly coloured and contain oils to attract and reward animals that disperse the seed. The structures offer niches for fungal contaminants that may be disseminated with the seed. Acacia seeds are often relatively large, ranging from several millimetres (e.g. *Acacia microbotrya*) to 1.5 cm in length (e.g. *A. stenophylla*), making them attractive food items for insects which may bore through the testa and remain within the seed during subsequent collection. In several parts of the world, including Australia and parts of Africa, indigenous people collect and trade seed as diet supplements. The explosive dehiscence of legume pods results in seed being deposited on the soil or vegetation surface, where contact with soil and litter mycoflora offers opportunity for contamination to occur.

Most acacia seed shows constitutive dormancy that must be broken by heat treatment or scarification. Boiling water treatment is effective, but may be uneven through a seed sample, therefore scarification within specially designed rotating drums or by the use of hot wires or even hydrochloric acid (to mimic the processing by animal gut) can give better control over germination rates. The boiling water and acid treatments however probably function as a partial sterilization treatment for microoganisms or microfauna borne superficially on the seed coat. The acid pretreatment is severe, having been used successfully with *A. mangium,* but is not suited to all species.

Movement of seed is by far the most common means of international germplasm diffusion (Table 1). The demand for seed of *A. mangium* has been very high over the past two decades, as major plantation estates based on this species have been established in Indonesia and Malaysia. There is growing interest in Vietnam, China and other humid tropical areas of South East Asia, and this trade in seed can be expected to continue for several years.

Cuttings and tissue cultured plantlets

Interest in movement of vegetatively propagated plants has been stimulated by the finding that some hybrids, for example, between *A. mangium* and *A. auriculiformis*, show rapid growth and improved silvicultural properties. Rooted cuttings are produced in the conventional way from young vigorously growing coppice shoots and all the commercially important species readily produce rooted cuttings. Some of the larger plantation companies are gradually moving to planting cuttings to take advantage of improvements in form and vigour of selected hybrids. Rooted cuttings pose the highest risk for movement of fungal pathogens and possible systemic agents such as viruses or phytoplasmas and their use should be reserved for exceptional circumstances, e.g. outstanding hybrids.

Tissue cultured propagules are derived from shoot meristems, which are surface sterilized to remove contaminants and grown axenically on sterile agar media. They offer a relatively safe way of moving vegetatively propagated trees but, as for cuttings, the risks and benefits of moving such material should be carefully weighed, as compared to seed of known provenance or pedigree. Tissue cultured hybrid plants have been moved between South East Asian countries for testing and incorporation in improvement programmes.

Pollen

Unlike some tree species there seems to be no significant movement of acacia pollen. Controlled pollination has proved to be difficult in improvement programmes to date, as the pollen is very difficult to collect and the small flowers are extremely difficult to isolate. The pollen is dispersed as polyads of multiple fused pollen grains, presenting challenges for collection and use in controlled pollination.

RECOMMENDATIONS FOR SAFE MOVEMENT OF ACACIA GERMPLASM

- Where possible, pest and weed risk analysis should precede movement of germplasm.

- Germplasm should undergo testing for presence of insect pests and pathogens and any recommended disinfestation treatment, to meet the requirements of a phytosanitary certificate, should be carried out before dispatch.

- Upon receipt, germplasm should be kept in isolation from other plants and grown under conditions conducive to symptom expression of recognized diseases.

- In exceptional cases where rooted material is moved, cuttings should be propagated in enclosed facilities, grown in sterilized media, transported in sealed containers and grown under quarantine for at least 6 months to allow symptom expression.

- Germplasm of all types should not be released into the field until confirmed to be free of insect pests and diseases. When this is not the case it should be destroyed.

- All packaging material used in the movement of germplasm should be destroyed.

TECHNICAL RECOMMENDATIONS

Note: All technical recommendations should be considered in addition to national and international phytosanitary measures.

Selection of the method of germplasm transfer should reflect the technical objectives of its use, such as broadening the genetic base of plantations, selecting disease-resistant provenances or families or for the purpose of introducing hybrids.

1. Seed

• Seed storage facilities should be cleaned and fumigated routinely.

• Seed for international movement should be collected from the tree rather than from the ground, as soon as adequately mature, to avoid contamination by pests and pathogens.

• Seed-lots for shipment should be thoroughly cleaned of trash, twigs and foliage fragments etc.

• Seed-lots should be fumigated with CO_2 to kill pests. This treatment will not affect pathogens so fungicidal treatment should be carried out just prior to sowing to avoid reducing viability.

• Boiling water pretreatment or acid scarification of seed should be used preferentially to assist with disinfestation.

2. Tissue cultured plantlets

• *In vitro* tissue should be derived from healthy plants, raised in closed facilities and grown on sterilized media.

• Plantlets should be cultured in axenic conditions free of contaminants.

• Plantlets should be shipped in sealed transparent containers and visually inspected before dispatch and after receipt. Unhealthy and contaminated cultures should be destroyed.

3. Rooted cuttings

• Movement of rooted cuttings is not recommended as several pathogens e.g. acacia rusts, of which there are many species, can inhabit symptomless plants.

INTERNATIONAL DISTRIBUTION OF GERMPLASM

Movement of germplasm should comply with the regulatory requirements of the recipient country and the export requirements of the donor country.

A description of tests, which have been carried out to assess the health of the germplasm, should accompany each shipment.

Re-export of the shipment should be accompanied by copies of the original phytosanitary documents, plus any additional actions taken during transit, which could affect the health of the consignment.

DETECTION AND TREATMENT

As indicated above, where germplasm is to be moved long distances, especially internationally, trees from which seed and vegetative material are to be derived should be as free as possible from pest infestations and diseases. Before issue of phytosanitary certificates, inspection of consignments, usually seed-lots, will normally be required. Treatment to minimize risks of spread of pests and diseases may also be required.

Detection of pests

Visual inspection of seed samples for the presence of pests may be combined with other quality-control measures, for example viability testing. This involves standard sampling protocols to ensure that samples are representative of the seed lots, and details are provided in International Seed Testing Association (1976).

Presence of pests can sometimes be determined by visual signs, including the presence of tunnels and larvae of Coleoptera (*Melanterius*) and Lepidoptera (*Cryptophlebia*) and associated frass. Seeds parasitized by Hymenoptera (*Bruchophagus*) are typically bloated proximally (due to the pupal cell) and slightly shrivelled distally. The testa of infested seeds is pale brown and not black as in healthy seeds. The above-mentioned insects and some others listed in Table 2 may also be detected visually in the seed-lots.

Treatment

Seed-lots collected by the ATSC are routinely treated with carbon dioxide to control insects. Seed is cleaned of all extraneous material and then sealed in laminated plastic bags containing carbon dioxide for a two-week period, prior to entering the storage areas. Seed brought into Australia from neighbouring countries is treated with methyl bromide. As noted above, the hot water and acid scarification methods of breaking dormancy are extra insurance against insect pest infestation although these measures may not be completely effective where insect larvae are sequestered within large seeds.

Detection of pathogens

Seed of acacias, in common with virtually all other crops and plantation species carries a complex mycoflora. Many of these fungi are present on the seed after harvest and colonize the testa and appendages during storage. These storage fungi are favoured by high humidity and temperatures between 15 and 30°C and if seed-lots are stored in such conditions there will be major reductions in viability. Other fungi, e.g. rusts, may be present in seed stands and may be actively sporulating during harvest. Seed, leaf

fragments and other trash could be contaminated with spores that remain in a dormant stage until susceptible host tissue is available for infection. Detection of these chance contaminants during any inspection is highly unlikely. The third category of seed-borne fungi includes those actively colonizing the seed coat, appendages or endosperm. These fungi may infect seedlings during germination. A list of pathogenic fungi detected in acacia seed is presented in Appendix I based on data published by Old and Yuan (2001).

Table 2. Some additional insects that feed on *Acacia* spp. and have the potential for transmission via germplasm.

Order: Family	Species	Natural range	Portion attacked and reference	Economic importance
Lepidoptera: Tortricidae	*Eucosma* spp.	Australia	Leaves, seed pods and/or flowers	Reduces flowering and seed set in Australia (van den Berg 1980a, 1982a)
Lepidoptera: Gelechiidae	*Xerometra crocina*	south-east and south-west Australia	Seed funicles and developing seeds	Minor seed predator in Australia (van de Berg 1980a)
Hemiptera: Scutelleridae	*Coleotichus costatus*	Australia	Developing and mature seeds	In Australia can caused seed sterility of >25% (van den Berg 1980b)
Hemiptera: Pentatomidae	*Dictyotus* spp.	Australia	Developing seeds through the pods	In Australia can caused seed sterility of >25% (van den Berg 1980b, 1982b)
Hemiptera: Cydnidae	*Adrisa* spp.	Australia	Seeds	Minor seed predator in (van den Berg 1980b, 1982b)
Hymenoptera: Pteromalidae	*Mesopolobus* sp.	Australia	Seeds	Causes seed mortality of <20% to 40% on *A. aneura* (Preece 1971)
Coleoptera: Curculionidae	*Diethusa* spp.	Australia	Developing seeds through the pods	In Australia causes losses in seed production of 5% to 13% (Auld 1983, 1986, van den Berg 1982c)

Detection may be by visual inspection or by plating seed on agar media, which promotes mycelial growth and sporulation of seed-borne fungi. Plating without surface sterilization will usually result in fast-growing storage fungi out-competing putative pathogens, so a surface sterilant, e.g. 5% sodium hypochlorite for a standard time (2 min may be suitable) followed by plating on a non-selective agar will yield colonies for identification (Yuan *et al.* 1989). In practice, few seed-lots given phytosanitary certificates are subjected to such rigorous testing for the presence of pathogens before dispatch.

Treatment

The CO_2 treatment, commonly used to disinfest seed pests, has virtually no therapeutic effect with regard to seed-borne fungi. Hot water treatment should greatly reduce superficial fungal contamination, however true seed-borne pathogens such as *Fusarium* species and many pigmented hyphomycetes that form resting propagules on or within seeds, would probably survive this treatment.

The boiling water treatment, necessary to promote seed germination, is probably the only measure widely used although surface sterilization with sodium hypochlorite prior to planting is a possibility, providing care is taken to maintain seed viability.

INSECTS WITH POTENTIAL PHYTOSANITARY IMPORTANCE FOR MOVEMENT IN GERMPLASM

Acacia mites

Aceria *spp. (Acari: Eriophyoidea: Eriophyidae)*

Significance

Colonies of these mites develop in young inflorescences of various Australian *Acacia* spp., causing bud tissue to proliferate and form dense witches' brooms instead of flower heads. They may also affect shoot tips and vegetative buds, in which they may survive undetected. Other so-far-undescribed species may form small hairy pinpoint patches containing mite colonies in small cavities on phyllodes (Fig. 1), or may occur as free-living or refuge-seeking vagrants on leaves, phyllodes, flower heads and young pods.

Fig. 1. *Aceria* sp. lesions on an *Acacia longifolia* phyllode.

Damage

Inflorescences may be grossly deformed, rendering them sterile, and probably acting as nutrient sinks. In heavy infestations, branches may be covered with, and weighed down by, large gall-like growths and seedlings may be stunted. Abortion of affected flowers and podlets may occur. Eriophyoid mites are also vectors of viruses and phytoplasmas (Lindquist *et al.* 1996).

Hosts

In Australia, only the mite species affecting *Acacia saligna, A. melanoxylon* and *A. falciformis* have been described. Various other *Acacia* species, including *A. dealbata, A. longifolia* and *A. mearnsii*, have been observed to display typical symptoms of eriophyoid damage (Neser pers. comm.).
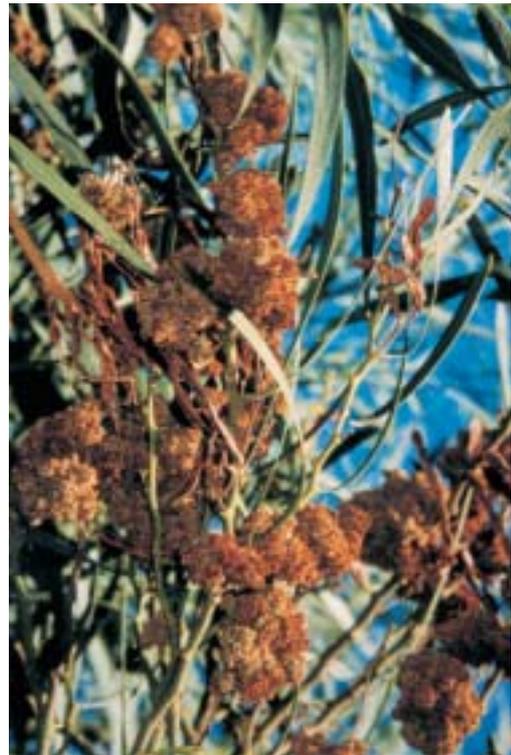

Fig. 2. Witches' broom on *Acacia saligna* caused by *Aceria acaciflorus.*

Geographical distribution
About 30 species of *Aceria* are known to have *Acacia* spp. as their host and they occur in South Africa, India, Indonesia, Egypt, the West Indies, Chile and Australia (Amrine Jr and Stasny 1994). The known Australian eriophyoids from *Acacia* occur in WA, SA, Victoria and NSW, but various species probably occur countrywide (Meyer 1990, Neser pers. comm.). There are possibly numerous undescribed species associated with different *Acacia* hosts in Australia, but no concerted survey or study of Australian species has been undertaken (Neser pers. comm.). Only three species had been described from Australia by 1990: *Aceria acaciflorus* from *Acacia saligna* in both western and eastern Australia and *Acacia melanoxylon* in the east, *Aceria falciformis* from *Acacia falciformis* (in NSW) and *Aceria burnleya* from *Acacia saligna* in Victoria, reportedly from blisters on the phyllodes (Meyer 1990).

Biology
There may be several generations a year. The life cycle of similar species may be as short as 7–12 days under favourable conditions, and some species have been shown to be vectors of damaging viruses and phytoplasmas of crops and ornamental plants. Single females may enter and survive in buds and are able to establish large colonies in suitable niches on susceptible plants. Unfertilized *Aceria* females produce male offspring that may then fertilize the female to allow production of female offspring (Lindquist *et al.* 1996). They disperse by wind or phoretically on insects and other animals visiting the host plants.

Detection
The presence of *Aceria* spp. can be inferred from the typical symptoms of distorted new growth and witches' brooms (Fig. 2). Single mites are not visible to the naked eye but may be detected by microscopic examination of affected host tissue such as dormant buds. They are worm-like, usually cigar-shaped with 4 legs at the cephalic end and are whitish to creamy pink. Symptoms may not be obvious for low populations of vagrant species. In some cases, symptoms may include yellow stippling, 'bronzing', 'rusting', 'silvering' or other discolouration of phyllodes. Vagrant species from African *Acacia* spp. have been recorded to cause abortion of buds, flowers and podlets.

Treatment
Destroy infested germplasm. Bud and graft material may contain minute eriophyoid mites and need to be treated chemically.

Acacia psyllids

Acizzia *spp. and* Psylla *spp. (Hemiptera: Psylloidea: Psyllidae)*

Significance

Accidentally introduced into a number of countries where it is considered a minor to serious pest, *Acizzia (Psylla) uncatoides* is currently the target of a biological control programme in Hawaii, where it is an important pest of the endemic *Acacia koa* and in France where it is a pest of the Australian species, *A. retinodes*.

Damage

The feeding of the nymphal stages causes most damage. Heavy psyllid populations cause chlorosis of leaves and tip die-back of new growth (Fig. 3). Young plants, supporting large populations, may suffer reduced growth and damage to meristematic

Fig. 3. Stunting of the growing tips caused by psyllids feeding on *Acacia mearnsii*.

tissue can result in plants having poor form. Nymphs and adults excrete honeydew, which provides a medium for the growth of sooty mould.

Hosts
Psyllids are generally narrowly host specific. For example, *Acizzia acaciaebaileyanae* has only been recorded from *A. baileyana* and *A. podalyriifolia* (Hodkinson and Hollis 1987) and *Psylla acaciaepycnanthae* is specific to *A. pycnantha* (Yen 1984). However *Acizzia uncatoides* is polyphagous. In California alone, it has been recorded from 58 of 112 species of *Acacia* and 3 of 6 species of *Albizia* examined (Munro 1965).

Geographical distribution
There has been some confusion in the literature about the relative status of *Psylla* and *Acizzia*, particularly with regard to the fauna on *Acacia* (Taylor 1999). Most of the Australian *Acizzia* were originally assigned to the holding genus *Psylla*. There are now thought to be some 30 *Acizzia* species distributed from Australia, New Zealand, the Old World tropics, North America, the Middle East and the Mediterranean area. The 22 described Australian species feed predominantly on *Acacia* but many more undescribed species are known. Of the 11 species of *Psylla* originally described by Froggatt, nine were collected from *Acacia* and four of these are currently ascribed to *Acizzia*. At least two species of acacia-feeding *Acizzia*, *Acizzia uncatoides* and *Acizzia acaciaebaileyanae*, have been introduced to New Zealand, South Africa, Italy France, North America, Hawaii, Mexico, Israel and England (Arzone and Vidano 1985; Bain *et al.* 1976; Capener 1970; Ferris and Klyvr 1932; Gagne 1971; Halperin 1986; Halstead 1992; Hodkinson and White 1981; Jensen 1957; Koehler *et al.* 1966; Leeper and Beardsley 1973; Leeper and Beardsley 1976; Madubunyi and Koehler 1974; Munro 1965; Pettey 1924; Rapisarda 1985; Tuthill 1943).

Biology
Eggs are laid either singly or in small groups on host plants. Favoured oviposition sites vary for different species but include stems of young shoots or the leaves. Nymphal instars are distinctly dorso-ventrally flattened with colour varying between species. Adults have wings, as opposed to the nymphal wing pads, which are held over the body like a pitched roof. Both adults and nymphs feed by sucking the plant juices. Favoured feeding positions vary between species, but include leaf and stem surfaces, and leaf axils. Adults can disperse short distances by flight and over longer distances on air currents.

The small immature stages of psyllids are often inconspicuous and could be transmitted by rootstock or cuttings.

Detection
Presence of insects on foliage and stems (Fig. 4) and characteristic damage such as chlorosis of leaves and tip die-back on new growth.

Treatment
Infected germplasm can be treated with a systemic insecticide. Surface sprays should not be used due to difficulties in ensuring effective application into leaf axils and other confined areas where early instar nymphs can hide. If there is doubt about the effectiveness of insecticide treatment the germplasm should be destroyed.

Fig. 4. Various stages of a psyllid, probably *Acizzia*, on *Dasineura* galls on *Acacia baileyana*.

Melanococcus albizziae (Maskell) (Hemiptera: Coccoidea: Pseudococcidae)

Significance
Sucks the sap of Australian acacias and can cause die-back of branches and death of trees.

Damage
This insect forms dense aggregations on the leaves, twigs and branches, often entirely covering twigs and branches for several centimetres. Damage is restricted initially to die-back of foliage on infested branches but can lead to branch death if infestations are not controlled (Fig. 5). Death of the entire tree may follow prolonged (several years) infestations of young or unhealthy trees. A large species complex of natural enemies is associated with *M. albizziae* in Australia and populations rarely develop to levels where damage is severe (Farrell 1985). Nymphs and adults also excrete honeydew, which provides a substrate for the growth of sooty mould.

Hosts
Melanococcus albizziae feeds on a wide range of Australian *Acacia* spp. Both bipinnate and phyllode species are utilized and host plants include commercially important species such as *A. melanoxylon* and *A. mearnsii.* Although originally collected from *Paraserianthes lophantha,* it has rarely been recollected from this host plant.

Fig. 5. Large colony of *Melanococcus albizziae* on the branches of *Acacia mearnsii.* Note the loss of foliage on some of the smaller branches.

Geographical distribution
The species is native to Australia and has been recorded from southern Queensland, NSW, ACT, Victoria and Tasmania (Williams 1985).

Biology
Adult females are wingless and sedentary. They are blue-black in colour, but also produce a white waxy secretion (Fig. 6), which partially covers the top of the insect and forms a brood sac beneath its body. Live young are laid into the brood sac with each female producing several hundred offspring. The reddish nymphs or 'crawlers' spend a few days to a few weeks in the brood sac before emerging to disperse over the plant. The crawlers are also the major dispersal phase of the species and use air currents to find new host plants. Crawlers and early nymphal stages tend to settle in sheltered locations on the plant, while later instars colonize the more exposed larger twigs and branches. All stages secrete a white waxy secretion. In early nymphal stages this secretion is powdery while in later stages it forms a series of transverse wax ridges on the top of

Fig. 6. Adult females of *Melanococcus albizziae* on the stem of *Acacia dealbata*.

the insect. Male *M. albizziae* occur in most populations but numbers are low and they are difficult to find.

In south-eastern Australia there are two to three generations per year and most stages of the insects can be found through out the year. While development continues over the winter months, females cannot sexually mature until spring. This results in reproduction of the summer generation being synchronized. Reproduction in subsequent generations is less synchronized.

This insect could be transported as small inconspicuous immature stages on rootstock or cuttings.

Detection
Presence of the insect, sooty mould or honeydew on foliage, twigs or branches.

Treatment
Destroy infested germplasm.

Phyllode spotting mirid bug

Rayieria *sp. (Heteroptera: Miroidea: Miridae)*

Significance

Nymphal and adult feeding may cause severe damage to the foliage of phyllodinous *Acacia* spp. When suitable phyllodinous hosts are rare, feeding can switch to bipinnate species. Severe damage can result in death of small or unhealthy plants.

Damage

Adults and nymphs of *Rayieria* sp. feed by inserting their stylets into the phyllode. A clear watery lesion of 2–4 mm diameter forms around the stylets within an hour of feeding commencement. The lesion dries out after the stylets are removed leaving a characteristic feeding spot (Fig. 7). Feeding damage results in host plants becoming brown (Fig. 8) and may lead to defoliation. In Australia *Rayieria* is a damaging pest to several phyllodinous acacias in eastern, western and central Australia (Neser pers. comm.).

Hosts

Host specificity testing in South Africa has shown that feeding is restricted to Australian *Acacia* spp. (Donnelly 1986). In Australia, *Rayieria* sp. has been recorded from *Acacia longifolia* and *A. rubida* in south-eastern Australia and *A. saligna* in Western Australia. However, host specificity trials in South Africa also showed it can feed on *A. mearnsii, A. cyclops, A. implexa* and *A. melanoxylon.*

Fig. 7. Characteristic *Rayieria* sp. feeding damage on a phyllode.

Geographical distribution
Native to Australia, the species has been collected from eastern, central and western Australia. While tested as a biological control agent in quarantine in South Africa, it was not subsequently released since one of its hosts was the commercially important species, *A. mearnsii* (Donnelly 1986).

Biology
Both sexes mate repeatedly and the female pre-oviposition period is about seven days. Females lay a total of about 80 eggs each. Groups of one to seven eggs are embedded in the stems of young host plants. Oviposition sites are covered with a waxy substance, from which characteristic respiration tubes or aeropyles protrude (Donnelly 1986). Eggs over-winter in cool to cold climates but do not have an obligate diapause phase. Under laboratory conditions hatching occurs approximately 21 days after oviposition. There are five nymphal stages lasting approximately three days each and adults can survive for four to eight weeks. Adults will fly actively if disturbed.

This species could be transmitted in root-stock or cuttings since groups of eggs are embedded in the young stems of host plants.

Detection
Presence of feeding spots on foliage or a white waxy substance covering eggs, which are embedded in young stems.

Treatment
As detection of the embedded eggs is difficult all germplasm should be destroyed if feeding damage is detected.

Fig. 8. Adult *Rayieria* sp. on damaged phyllode of *Acacia longifolia.*

Acacia galling midges

Dasineura *spp. and* Asphondylia *spp. (Diptera: Cecidomyiidae)*

Significance

Larvae form galls in flower buds, flowers, young pods, as well in vegetative tissue, depending on species (Fig. 9). Damage to reproductive tissue can totally suppress seed production. *Asphondylia* spp. may have obligate associations with one or more fungi, including pathogens (Gagné 1989; Adair *et al.* 2000; Kolesik 2000).

Damage

Affected flower buds, flowers, podlets or developing seeds do not mature, and seed production may be almost totally suppressed. Larger galls formed by some species of *Dasineura* act as nutrient sinks. Some species cause galls, or stunt vegetative tissue such as shoot-tips and phyllodes. This group of midges includes some important crop pests found in most parts of the world, but the host ranges of the phytophagous species are generally limited, in many cases restricted to a single host species.

Fig. 9. Healthy pods of *Acacia cyclops* next to *Dasineura dielsi* galled pods.

Hosts

Most *Acacia* spp., if not all, may be hosts to cecidomyiids which invade their reproductive parts, and many have cecidomyiid galls on phyllodes and twigs. A complex of different species may occur on a single host species: *Acacia mearnsii* is known to be a host of at least 8 different cecidomyiid species in immature reproductive parts (Adair 2000). Some midges alternate between different species of hosts. *A. baileyana* often has small, colourful, spherical galls on the leaflets. Adair *et al.* (2000) lists some 50 Australian *Acacia* spp. that are hosts to *Dasineura* spp. and *Asphondylia* spp.

Geographical distribution

Although widespread in Australia, only two of the many species associated with Australian *Acacia* spp. have been described: *Dasineura acaciaelongifoliae* (Skuse 1890) and *D. dielsi* (Rübsaamen 1916). There are various undescribed species and some may prove to belong to other genera. New taxonomic studies are now in progress (P. Kolesik pers. comm.).
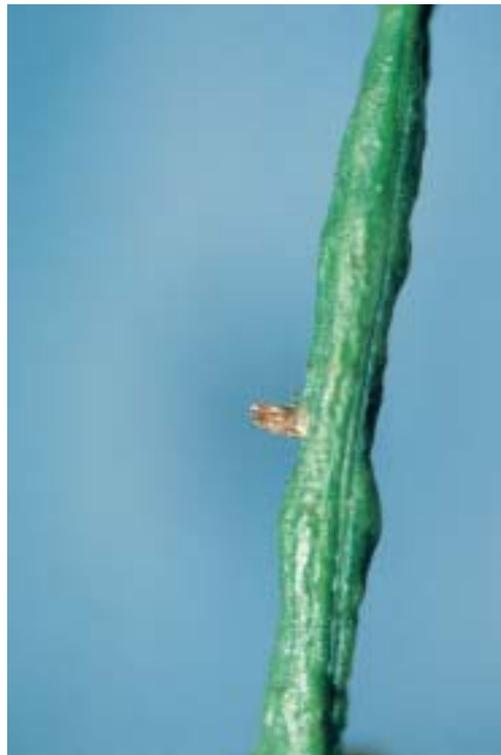

Fig. 10. Pupal cast of *Asphondylia* sp. protruding from an Acacia longifolia pod.

Biology
Depending on the species, there may be a single generation per year with an extended larval diapause, two generations that utilize different host parts for development, or various generations using different *Acacia* host species in succession. Eggs are minute and hardly visible to the naked eye. They are usually inserted into the immature flower heads, buds or flowers or placed on the ovaries and young pods, depending on species. Newly hatched larvae enter the host tissue or induce host tissue proliferation to eventually enclose them. *Asphondylia* spp. are typically solitary (Fig. 10), whereas *Dasineura* spp. may be gregarious within an organ. Larvae are whitish to yellow or reddish. Some species have rapid successive generations, some diapause as larvae and pupate in the affected host tissue, and larvae of others fall to the ground and form delicate white cocoons in the leaf litter or soil where they diapause and later pupate. The adult midges are usually only 1.5–2.5 mm long. They are very mobile and may disperse widely, but are short-lived, surviving at most for a few days.

In their native habitat, the majority of larvae and pupae may succumb to a variety of hymenopterous parasitoids, predatory or parasitic mites and other predators, but the midges are sufficiently prolific and mobile to successfully affect even isolated host plants and to maintain high levels of sterilization of hosts.

Dispersal of adults may be aided largely by air currents and to a lesser extent by movement of affected growing plants or plant parts. Young larvae require living, developing host tissue to be able to mature.

Detection
Careful examination of inflorescences, green pods and vegetative tissue for signs of atypical organ development or galls.

Treatment
Destroy infested germplasm.

Melanterius spp. (Coleoptera: Curculionoidea: Curculionidae)

Significance
Feeding by adults and larvae on ripening seeds either destroys seeds outright or causes infertility, reducing seed production.

Damage
In Australia seed mortality due to *Melanterius* spp. is variable. New (1983) found that the level of damage in successive years for the same host plant species could range from 0% to 89%. In South Africa, species such as *M. ventralis* and *M. acaciae*, which are now well established as biological control agents, are causing seed damage in the order of 90% to 100% at some localities.

Hosts
Melanterius spp. are restricted to Australian acacias and their close relative *Paraserianthes lophantha.* The genus is thought to have evolved to utilize a range of acacias. Twenty out of twenty-four *Acacia* spp. examined in and around Sydney were found to have *Melanterius* spp. as seed predators, as did a further eight *Acacia* spp. in arid and semiarid areas (Auld 1991). The host specificity of *Melanterius* is variable with individual species not necessarily specific to a single species of *Acacia*.

Geographical distribution
There are some 88 species of *Melanterius* described from Australia, while only two species have been recorded from elsewhere (Papua and New Caledonia). The genus has been studied mainly in temperate Australia, and *Melanterius* has been found on most acacias examined. Several of the Australian species have been introduced into South Africa for

Fig. 11. Adult *Melanterius maculatus.*

the biological control of seeds of Australian acacias. These are *M. ventralis* for control of *A. longifolia, M. acaciae* for control of *A. melanoxylon, M. servulus* (strain A and B) for control of *A. cyclops* and *P. lophantha,* respectively, and *Melanterius* sp. (near *maculatus*) (Fig. 11), for the control of seeds of *A. dealbata* (Dennill and Donnelly 1991; Dennill *et al.* 1999) and *A. mearnsii.*

Biology
All species of the genus *Melanterius* have similar biology. There is one generation per year with adults and larvae feeding on the green developing seeds of their hosts. In the Southern Hemisphere, inactive, reproductively immature adults are found from mid summer (January) to late winter (August). Mating is synchronized with the period of flowering and pod development of the host plant. Oviposition occurs either directly into the pods or into the seeds, or onto the pod surface. The larvae tunnel into the green ripe seeds, (Fig. 12) consuming the entire seed and leaving only the seed coat. Usually only one larva develops per seed and development is timed to span the development of the seed. Pupation occurs in the soil with adult emergence occurring 6–8 weeks later.

Detection
Presence of tunnels and larvae in seeds; mucilage exudations and emergence holes of larvae in pod walls.

Treatment
Destroy the germplasm.

Fig. 12. *Melanterius servulus* larva in an *Acacia cyclops* seed.

Macadamia nutborer

Cryptophlebia ombrodelta *(Lower) (Lepidoptera: Tortricoidea: Tortricidae)*

Significance
Macadamia nutborer (Fig. 13) is capable of heavy infestation of *Acacia* spp., as well as being a pest of several commercial species including macadamia, lychee and tamarind.

Damage
Larvae burrow into young fruit or seed pods, either feeding on the kernel or seeds. For *Acacia* spp. larvae may destroy all seeds in an infested pod. For commercial host species, feeding damage may either destroy the fruit, cause premature seed fall, or the tunnel created by the burrowing larvae may provide access for secondary infestations by other insects or pathogens (Ironside 1974). In some hosts, tunnelling of young shoots has been reported.

Fig. 13. Adult female of *Cryptophlebia ombrodelta*.

Hosts
Larvae feed on *Acacia* species with large pods (Common 1990). Recorded hosts include *A. farnesiana* (Froggatt 1897), *A. cyclops* (van den Berg 1980) and *Acacia* spp. (Bradley 1953; Davis 1962). Larvae are also serious pests of macadamia (Macadamia integrifolia, Proteaceae), lychee (*Litchi chinensis*, Sapindaceae) and tamarind (*Tamarindus indica, Caesalpiniaceae*) (Ironside 1974; Lingappa and Siddappaji 1981; Morton 1987). Other hosts include *Adenanthera pavonina* (Mimosaceae), *Sesbania* (Fabaceae), *Bauhinia*, *Cassia* with large pods, *Poinciana pulcherrima*, *Delonix regia* (all Caesalpiniaceae), *Cupandiopis anacardoides* (Sapindaceae), and *Buckinghamia celsisima* (Proteaceae) (Common 1990). For a full list of host species see Ironside (1974).

Geographical distribution
Originally described from Sydney (Common 1990), the species has also been recorded from Queensland and the Northern Territory (Davis 1962). It is also widely distributed outside of Australia and can be found from India and Sri Lanka to Taiwan and from the Philippines to Australia, the Solomon Islands and Hawaii (Commonwealth Institute of Entomology 1976; Common 1990).

Biology
Eggs are laid exterior to the seed or pod. On *Acacia* spp., the newly hatched larvae tunnel into the pod where they feed on the seeds. Larvae are a uniform dull yellowish brown, but sometimes have a greenish tint. Pupation occurs in a silk cocoon within the pod. Before pupation, the hole through which the larvae entered the pod is enlarged and the head of the cocoon is orientated towards it. The adult moth exits the pod through this hole.

Detection
Larvae can be detected in seed pods by the presence of holes in the pods, often with protruding frass. Larvae feeding on seeds excised from pods can be detected by frass emanating from damaged seeds.

Treatment
Destroy infested seeds. Adult moths can be trapped with commercially available pheromone traps.

Bruchophagus spp. (Hymenoptera: Chalcidoidea: Eurytomidae)

Significance
Larvae develop in the green seeds of various Australian *Acacia* spp. (Fig. 14). Diapausing larvae remain inside affected seeds for many months and under certain conditions may even diapause for a number of years (such as in packets of seeds). One species, *Bruchophagus acaciae,* has become established in the South Island of New Zealand, mainly on *A. dealbata, A. baileyana* and *A. decurrens* (Hill *et al.* 2000). The exact identity of specimens recently found there, in seeds of *A. rubida, A fimbriata, A. myrtifolia, A. deanei, A. retinodes* and *A. caesiella*, and in seeds of *A. dealbata* from the North Island, has not yet been confirmed (Neser pers. comm.).

Damage
Feeding larvae make neat round cavities in seeds. In Australia seed mortality ranges from 5 to 24% (Kluge 1989); in New Zealand seed damage in excess of 80% (89% in *A. silvestris*) has been reported (Hill *et al.* 2000).

Fig. 14. Pod of *Acacia longifolia* cut open to show *Bruchophagus* sp. larvae in the seeds.

Hosts
In Australia the *Acacia*-feeding *Bruchophagus* spp. has been recorded from *A. cyclops* (van den Berg 1980; Kluge 1989), *A. saligna* (van den Berg 1980), *A. floribunda* (S. Neser, pers. comm. in (Kluge 1989)), *A. sophorae* and *A. elongata* (Auld 1986; Kluge 1989) and *A. dealbata, A. longifolia* and *A. mearnsii* (van den Berg 1982; Auld 1986; Kluge 1989). *Bruchophagus acaciae* was originally described from *A. dealbata* in New Zealand, although it has recently been reared from *A. mearnsii* in Victoria, Australia. Seeds of 16 species of *Acacia* (10 of which have not previously been recorded as hosts) affected by larvae of as yet unidentified *Bruchophagus* species were recently collected in eastern Australia between Canberra and Sydney, and from *A. genistifolia, A. dodonaeifolia, A. dealbata* and *A. mearnsii* in Tasmania, from *A. pycnantha* in South Australia and from *Acacia* sp. cf. *strongylophylla* in the Northern Territory (Neser pers. comm.).

Geographical distribution
The known *Acacia* seed-feeding *Bruchophagus* are native to Australia. At least *Bruchophagus acaciae* must have been accidentally introduced into New Zealand, probably around a hundred years ago. *Bruchophagus* spp. are also currently being evaluated for introduction into South Africa as alternative biological control agents of seeds of different Australian acacias and the specimens reared from seeds are being subjected to a taxonomic study (Prinsloo *et al.* 2001).

The genus *Bruchophagus* also contains species that have been recorded from commercially important plant species (Kluge 1989). Host records of Australian specimens from early in the previous century are non-existent or in need of confirmation. Within Australia, a number of species previously placed in the genus *Eurytoma* belong to *Bruchophagus* (Bouček and Brough 1985). Some of the *Eurytoma* species which van den Berg collected and attributed as parasites of *Melanterius* (van den Berg 1980a; van den Berg 1982) were later identified as *Bruchophagus* spp. and as *Risbecoma* sp.—a closely related genus of wasps also known elsewhere from seeds of legumes (Neser pers. comm.).

Biology
There is one generation a year and larvae go through an obligatory diapause of one to several years. Adults are only found for a short period corresponding to the flowering period of the host plant (actually the period when suitable young pods are present). In captivity, females on average produced about 40 offspring (Kluge 1989); dissections of ovaries showed a potential of three to four times this number in *B. acaciae* (Hill *et al.* 2000). Pods are suitable for oviposition when they are between 1 and 4 mm in width or from the time when the cotyledon first becomes visible until it has filled the seed cavity. On completing feeding, the larva creates a diapause cell within the seed by lining the inside of the feeding cavity with a black secretion. Only one larva is found per seed. Seeds containing diapausing larvae drop from the tree along with healthy seeds or at least temporarily adhere to the dry pod walls. Pupation occurs just prior to emergence.

Detection
Parasitized seeds of *A. sophorae* are typically bloated proximally (due to the pupal cell) and slightly shrivelled distally. The testa of infested seeds is pale brown and not black like healthy seeds (Fig. 15). Larvae of some species puncture and again block the testa of the seed at one, or two places, according to the species, and the seed is glued to the pod wall at the puncture site(s), so that affected seeds remain attached to pod remains on the ground.

Treatment
Destroyed infested germplasm.

Fig. 15. Healthy *Acacia longifolia* seed next to seed being parasitized by a *Bruchophagus* sp. larva. Note the difference in colour (the healthy seed is black) and the frass associated with the parasitized seed.

Trichilogaster spp. (Hymenoptera: Chalcidoidea: Pteromalidae)

Significance
The larvae form galls within the flower buds (and occasionally in vegetative tissue) of Australian *Acacia* species (Figs 16 and 17) resulting in reduced seed production, reduced biomass and in some cases tree mortality. Some species develop entirely in vegetative tissue of the host *Acacia*.

Damage
Gall formation may cause up to 100% reduction in seed production. In Australia such levels of damage are rarely attained due to the incidence of parasitism and competition. In South Africa established populations of *T. acaciaelongifoliae* cause reductions in seed production of 85–100%, reduce plant biomass by enhancing phyllode abscission and may cause tree mortality (Dennill *et al.* 1999).
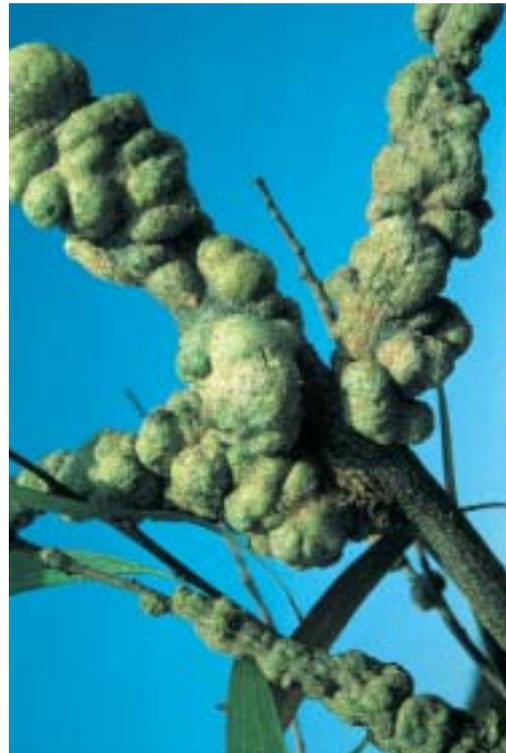

Fig. 16 (top). Example of shoot galling of *Acacia maidenii* by *Trichilogaster maideni*.

Fig. 17 (bottom). A sectioned *Trichilogaster trilineata* gall from *Acacia longifolia* showing larvae and a pupa.

Hosts
There are at least 10 species of *Trichilogaster* and all form stem or flower galls on Australian *Acacia* species. In Australia *Trichilogaster acaciaelongifoliae* has been reared from *A. longifolia* and the closely related *A. floribunda*. However in South Africa it has on rare incidences 'expanded' its host range to include *A. melanoxylon* and *Paraserianthes lophantha*. An unidentified species, *Trichilogaster* sp., has been reared from *A. pycnantha*. *Trichilogaster trilineata* forms galls in vegetative tissue and inflorescences of *A. mearnsii, A. dealbata and A. baileyana* (Neser pers. comm.).

Geographical distribution
The genus is endemic to Australia. Two species, *T. acaciaelongifoliae* and *Trichilogaster* sp., collected from populations in NSW, South Australia and Victoria, have been introduced into South Africa for the biological control of *A. longifolia* and *A. pycnantha,* respectively.

Biology
There is one generation per year and reproduction in *T. acaciaelongifoliae* is parthenogenetic, although variable but small proportions of males do occur in the population. The female wasp lays one to several eggs in the immature bud of their host plant in mid summer. Eggs remain dormant until the following spring when they hatch as the bud starts to enlarge. As the larvae develop the buds become distorted and develop into substantial galls containing isolated chambers each bearing a wasp larva. Galling of a reproductive bud completely prevents the development of that inflorescence, and the gall acts as a nutrient sink, that generally prevents seed formation on the affected branch. This insect can be transported on rooted stock or cuttings.

Detection
Presence of galls in flowers and buds. These may not develop until several months after oviposition.

Treatment
Remove galls from infested germplasm. Destroy the excised galls.

DISEASES WITH POTENTIAL PHYTOSANITARY IMPORTANCE FOR MOVEMENT IN ACACIA GERMPLASM

Gall rusts

Causal organisms
Uromycladium tepperianum (Sacc.) McAlpine, *U. notabile* (Ludw.) McAlpine. *U robinsonii* McAlpine.

Significance
There are at least seven known species of *Uromycladium* on acacias (McAlpine 1906). Three of these are commonly associated with significant damage associated with gall formation on plantation acacias, including a report of *U. robinsonii* causing die-back of terminal shoots of *Acacia melanoxylon* in New Zealand (Dick 1985).

The most important rust pathogens however are *U. tepperianum* and *U. notabile*. In 1987, the South African government approved the release of inoculum of *U. tepperianum* in an attempt to achieve biological control of *A. saligna*, an introduced Australian species that has become a major weed problem. The pathogen is now well established in South Africa (De Selincourt 1992).

Symptoms and signs
The most characteristic symptom of infection with *Uromycladium* spp. is the formation of reddish-brown, globose or irregularly shaped galls several centimetres in diameter on stems and shoots (Figs 18 and 19).

Fig. 18 (top). Globose gall caused by *Uromycladium notabile* on *Acacia mearnsii*.

Fig. 19 (bottom). Branching gall caused by *U. tepperianum* on native *Acacia* sp.

Inflorescences, phyllodes, fruits and shoot tips can also be infected causing gross malformation (Gathe 1971). Fresh galls are covered with powdery masses of spores. Older galls of both pathogens become dark brown and eventually blackened and are commonly invaded by tunneling insects. Susceptible trees may bear large numbers of branch galls (Fig. 20) which can affect the form of the tree due to shoot girdling and repeated branching (Fig. 21). Severely infected trees may die.

Hosts
In Australia these rusts occur on a very wide range of *Acacia* spp. with at least 100 species of acacia being recorded as hosts of *U. tepperianum* (McAlpine 1905; Warcup and Talbot 1981; Gathe 1971). The fungus also infects *Paraserianthes lopantha* ssp. *lopantha* (Willd.) Nielson in Australia and *P. lopantha* ssp. *montana* (Junghuhn.) Nielsen in Java. *U. notabile* is extremely common in southern Australia on a wide range of hosts in native forests, woodlands and plantations of bipinnate acacias such as *A. mearnsii* and *A. dealbata*. Morris (1987) showed that native African *Acacia* spp. are not susceptible to *U. tepperianum*.

Geographical distribution
These rust fungi are widely distributed in Australia on a great number of *Acacia* spp. and have spread to New Zealand. *U. tepperianum* occurs in Java on *P. lopantha* ssp. montana (McAlpine 1906) and in Papua New Guinea on *Albizia* (*Paraserianthes*) spp. (Shaw 1984). Undetermined species of *Uromycladium* have also been reported on *Albizia* (*Paraserianthes*) *falcataria* in Sabah and the Philippines (de Guzman *et al.* 1991). As noted above

Fig. 20 (left). *Acacia pycnantha* heavily infected by *U. notabile.*

Fig. 21 (right). *Acacia* sp. heavily infected by *U. tepperianum*. Note witches broom symptom.

U. tepperianum has been introduced into South Africa as a biocontrol agent and has proved highly effective (Morris 1997).

Biology and transmission
Infective spores are wind blown and can infect virtually any plant part except older stems with thick bark. The host reacts to infection by producing the characteristic gall tissue. The rust completes the whole life cycle on a single tree and during the growing season a gall of moderate size will produce many millions of spores that can infect neighbouring trees or disperse the fungus over longer distances. Galls can persist for several seasons forming new sporulating tissue in successive years. The life cycles of *Uromycladium* species are complex. *U. notabile* produces orange urediniospores on fresh galls (Fig. 23b,c) followed by darker chocolate brown teliospores. *U. tepperianum* however, only forms teliospores. *U. robinsonii* forms both spore types, usually on phyllodes, but can also induce cankers on twigs and small branches.

Detection
The rusts can be distinguished by gall morphology, the presence or absence of uredinia and the surface ornamentation of teliospores (McAlpine 1906). Teliospores of *U. tepperianum* bear converging longitudinal striations (Fig. 22) whereas the warty ornamentation of teliospores of *U. notabile* are roughly arranged in latiduninal rows (Fig. 23a).

Fig. 22. *Uromycladium tepperianum*. Striations on teliospore surface converge toward apex.

Treatment
No eradicative treatment for these rusts has been attempted with acacia germplasm.

Fig. 23 (a) (right) *Uromycladium notabile.* Warts on teliospores surface are arranged in rows. **(b) (below left)** *U. notabile* urediniospores, arrow shows germ pores. **(c) (below right)** *U. notabile* urediniospores, showing ornamentation of spores.

Uromycladium leaf rust

Causal organism
Uromycladium alpinum McAlp.

Significance
Unlike the other *Uromycladium* spp. inclu-
ded in this manual, *Uromycladium alpinum*
infection results in pinnule drop, especial-
ly of lower leaves (Fig. 24). In some reports
from South Africa, defoliation by this fun-
gus has been described as severe (Morris *et
al*. 1988).

Symptoms and signs
Uromycladium alpinum infection typically
results in the formation of single, small cir-
cular leaf spots on the pinnules of infected
leaves (Fig. 25). Uredinia can be found on
both surfaces of pinnules and also on
branches and stems. Spots bearing ure-
dinial sori are commonly surrounded by

Fig. 24 (top). Infected branch with loss of pinnules.

Fig. 25 (bottom). *Uromycladium alpinum*. Minute
lesion on pinnule of *A. mearnsii*.

yellow halos. Uredinia are often first seen as small raised blisters that rupture to expose the reddish-brown spore masses (Morris *et al.* 1988) (Fig. 26).

Hosts
Uromycladium alpinum has been reported from a number of Australian *Acacia* species including, *A. mearnsii* (Gibson 1975; Morris *et al.* 1988; Wingfield and Roux 2000), *A. dealbata* (Browne 1968; Bakshi 1976; Dick 1985), *A. buxifolia, A. dallachiana, A. implexa, A. linifolia, A. decora, A. spectabilis* and *A. terminalis* (Morris *et al.* 1988).

Geographical distribution
Uromycladium alpinum has been reported only from Australia (McAlpine 1906; Bakshi 1976; Dick 1985), where it is indigenous and South Africa (Morris *et al.* 1988) where it is likely to be an exotic introduction.

Biology and transmission
There is little published information available on this pathogen and its impacts on plantation acacias. Uredinia produce large numbers of spores which are adapted for airborne dispersal.

Detection
Uromycladium alpinum can be detected by the presence of circular spots on pinnules and stems on which the rust sporulates.

Treatment
No treatment is available or warranted.

Fig. 26. Urediniospores of *U. alpinum* note equatorial germ-pores (light circular patches).

Phyllode rust

Causal organism

Atelocauda digitata (G.Wint.) Cummins and Y. Hiratsuka syn. *Uromyces digitatus* Winter, *Uromyces phyllodiorum* B. and Br.) McAlp., *Racospermyces digitatus* (G.Wint.) J. Walker *comb. nov.*

Significance

A. digitata has come to prominence as an important pathogen on plantation-grown acacias only over the last few years (Old 1998). The genus has been recently revised (Walker 2001) and those species on *Acacia* have been renamed as *Racospermyces* spp. The disease occurs on native acacia species in many parts of Australia (McAlpine 1905) and is now known to be common in Indonesia, in the major plantation-growing areas of Sumatra and Kalimantan, especially on *A. mangium,* the most widely grown species.

Symptoms and signs

Infected phyllodes, shoot tips, petioles and even fruits may suffer gross malformation (Fig. 27), or more orderly cell proliferation in the form of galls or bullate (blister-like) swellings. These blisters are cinnamon brown in colour and are covered with spores when young, but darken later to become almost black (Fig. 28). The surface of older lesions is often colonized by fungal parasites and rust spores may be sparse.

Fig. 27 (left). Apical meristem of *Acacia mangium,* malformation caused by *Atelocauda digitata (Racospermyces digitatus)* infection.

Fig. 28 (right). Typical bullate swellings of *A. mangium* phyllode with sporulating *A. digitata.*

Fig. 29 (top). The crown of *A. mangium* sapling extensively infected by *A. digitata.*

Fig. 30 (bottom). Seedling in nursery infected by *A. digitata.*

Trees in young stands of *A. mangium* can be heavily infected (Fig. 29). The disease also occurs in nurseries (Fig. 30) where all stock can be infected and must be destroyed. Failure to destroy seedlings results in the pathogen being introduced into newly planted areas. In plantations young trees may be highly susceptible to damage as apical shoots are malformed and trees become multi-stemmed. Foliage of older trees bearing many rust pustules may be prematurely shed. Light infections may have little effect on growth but such trees act as sources of inoculum for young plantations.

Hosts
The rust has been found on a wide range of tropical and sub tropical acacias including *Acacia aulacocarpa, A. auriculiformis, A. crassicarpa, A. koa, A. leptocarpa, A. mearnsii, A. mangium, A. polystacha.*

Geographical distribution
Australia (northern, eastern and southeastern), Papua New Guinea, China, Hawaii,

Indonesia (including Java, Sumatra and Kalimantan) and New Zealand. Despite the presence of large areas of tropical acacia plantations in Malaysia, Thailand, Vietnam and India, the rust has not so far been formally recorded in these countries.

Biology and transmission
New infections appear late in the rainy season and become more evident over the next few months as infected phyllodes become distorted (Fig. 28) and bear masses of pustules. Phyllodes infected by *A. digitata* show a range of symptoms depending on the stage of the life cycle that is present.

Although all 5 spore stages have been reported, (McAlpine 1905; Gardner and Hodges 1985; Walker 2001) indicating that the rust is potentially macrocyclic, on some hosts e.g. *A. mangium* in northern Queensland and Indonesia the uredinial stage appears to be absent. The large bullate lesions formed on this host have been found to simultaneously bear both aecidiospores and teliospores (Figs 31–33). On other hosts, e.g. native

Fig. 31. Sporulating lesion of *A.digitata* on *Acacia celsa*, this lesion bore both aecidiospores and teliospores (see Figs 32 and 33).

A. auriculiformis and *A. crassicarpa* in northern Queensland (Old unpublished) pustules with very little tissue hypertrophy can be found. These may be uredinia but this remains to be confirmed as urediniospores and aecidiospores are morphologically similar. In his revision of the genus, Walker (2001) suggested that the new taxon, *Racospermyces digitatus*, is likely to encompass a complex of closely related acacia rusts. In common with other rust pathogens, spores are airborne and juvenile tissue is especially susceptible to infection. The fungus is not seed-borne, however the vast number of spores produced on lesions, and the capacity of this fungus to infect flowers and fruits makes chance contamination of seed-lots a possibility.

Detection
Identification rests on the presence on bullate lesions of the distinctive teliospores that bear finger-like (digitate) protrusions at their apices (Fig. 33). Aecidiospores also occur on these lesions but can also be found on smaller eruptions, which develop from sub epidermal pycnia in advance of gross tissue deformity. Aecidiospores are thick walled and highly ornamented with several prominent equatorial germ pores (Fig. 32).

Treatment
No eradicative treatment for these rusts has been attempted with acacia germplasm.

Fig. 32 (top). Aecidiospores of *A. digitata* showing equatorial germ-pores.

Fig. 33 (bottom). Teliospores of *A. digitata* showing apical finger-like protrusions.

Botryosphaeria canker

Causal organisms
Botryosphaeria spp., of which the most common are *B. rhodina* (Cooke) v. Arx, anamorph *Lasiodiplodia theobromae* (Pat.) Griff. & Maubl.(= *Botryodiplodia theobromae* Pat.) and *B. dothidea* (Moug.Fr.) Ces.& de Not. anamorph *Fusicoccum aesculi* Corda (=*Dothiorella aesculi* Petrak).

Significance
Impacts of these pathogens are limited in vigorous, well-managed stands, where they are found mainly on suppressed trees. If stands are stressed, for example where species or provenances are poorly adapted to climatic and edaphic factors, especially drought, damage can be extensive. Severe cankers were reported by Pongpanich (1997) on *A. auriculiformis*, associated with infection by a *Botryosphaeria* sp. in a trial in western Thailand with up to 80% mortality in some seed-lots. A replicate trial in a higher rainfall region in eastern Thailand showed only minor crown damage. Provenance trials of this species have also been severely damaged by *Botryosphaeria* spp. in Kalimantan (Indonesia) and in a moderately dry region of north east Sri Lanka (Figs 34 and 35). In Kwazulu Natal in South Africa, *A. mearnsii* damaged by hail or wind can suffer extensive stem cankers induced by *Botryosphaeria* infection.

Fig. 34 (left). Stand of *Acacia auriculiformis* in Sri Lanka affected by *Botryosphaeria* canker.

Fig. 35 (right). Detail of infected tree showing elongated canker and black fruiting structures.
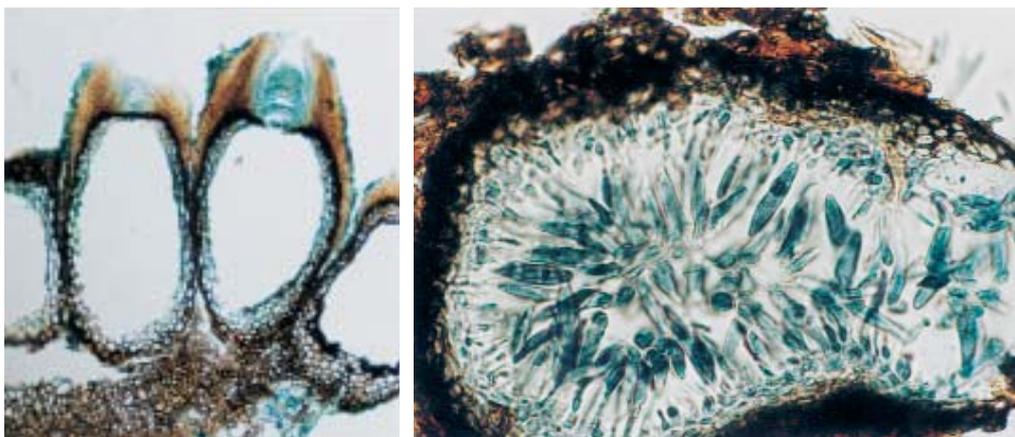
Symptoms and signs

Symptoms can vary from minor stem lesions to very large diffuse girdling cankers (Fig. 35) that can kill the main stem, resulting in multiple branching and loss of form (Fig. 34). Infection also weakens stems and causes breakage and dead tops. Cankers are darkly discolored (Fig. 35) and may be cracked especially toward the centre of the lesions. Fruiting bodies of *Botryosphaeria* spp. or their anamorphs can usually be found on the cankers themselves, especially at margins between diseased and healthy bark or on newly dead branches. The fruiting bodies are spheroidal, darkly pigmented and partially submerged in the outer bark but can be readily seen through a hand lens (Figs 36 and 37). If perithecia or pycnidia are sliced through with a scalpel or razor the contents appear, shining white. Microscopical examination of these contents shows spores in all stages of development including the characteristic asci and ascospores (Fig. 38).
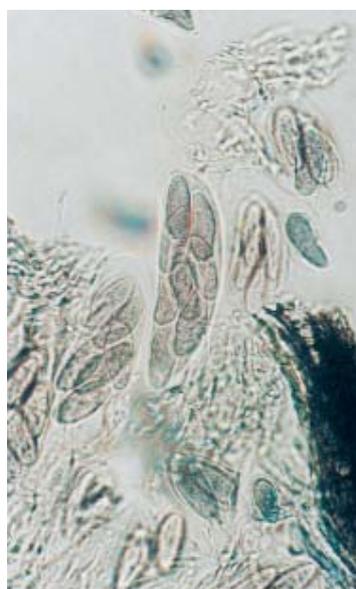

Fig. 36 (top-left). Longitudinal section of perithecia (sexual fruiting bodies) of *Botryosphaeria* sp. (K. Pongpanich)

Fig. 37 (top-right). *Fusicoccum* (asexual stage) of the fungus within black fruiting body. (K. Pongpanich)

Fig. 38 (right). Asci and ascospores of *Botryosphaeria* sp. (K. Pongpanich)

Hosts
A wide range of woody plants including horticulturally important plants and forest trees are attacked by *Botryosphaeria* spp. For plantation acacia species records of damage are most frequent on *A. auriculiformis, A. crassicarpa* and *A. aulacocarpa* in the tropics and on bipinnate temperate species such as *A. mearnsi* and *A. melanoxylon*).

Geographical distribution
These fungi appear to be worldwide in distribution, for example *Lasiodiplodia theobromae* is found throughout the tropics. There are reports of damage to plantation acacias from Thailand, Indonesia, Sri Lanka, India, Australia (Old *et al.* 2000), New Zealand and South Africa (Roux *et al.* 1997). As the interest in acacias as a short rotation plantation crop in the tropics continues, further reports will undoubtedly occur.

Biology and transmission
Generally regarded as wound pathogens, there is evidence that *Botryosphaeria* spp. are commonly present in symptomless tissue as latent infections, possibly originating through foliar invasion. The fungi are, therefore, likely to be present in all stands at a low level, and vigorous trees can be infected but show no symptoms in the absence of environmental stress (Smith *et al.* 1996).

When trees are stressed by drought, insect defoliation, hail or wind damage, diffuse cankers can rapidly develop. These infections kill cambial tissue and sapwood but do not cause decay. Open lesions exposed by death of bark and cambium, however, can act as avenues for infection by decay fungi. The vast numbers of fruiting bodies on infected stems persist and disperse viable spores long after the substrate is killed and chance contamination of plantation products, including seed pods and seed is quite likely.

Detection
The profuse sporulation and characteristic conidia and ascospores make it possible to identify these pathogens to genus by direct microscopical examination. If stem material is dry, then small pieces of bark bearing fruiting bodies can be incubated in moist chambers to promote sporulation. Isolation from freshly infected stems using standard mycological culture media is also effective. Isolates vary in readiness to sporulate in culture and exposure to near ultra-violet light may be needed.

Treatment
No eradicative treatment available.

Cylindrocladium foliar blight

Causal organisms
Cylindrocladium spp., including *C. crotolariae* (Loos) Bell & Sobers, *C. candelabrum* Viegas, *C. ilicicola* (Hawley) Boedijn & Reitsma, *C. quinqueseptatum* Boedijn & Reitsma, syn. *C. reteaudii* (Bugn.) Boesew. *C. scoparium* Morgan and *C. theae* (Petch) Subramanian.

Significance
Cylindrocladium spp. are very damaging on some hardwood plantation species, especially in the tropics and subtropics. For example plantations of several widely grown species, including *Eucalyptus camaldulensis*, *E. tereticornis* and *E. urophylla*, can be severely affected by leaf blight caused by *C. quinqueseptatum*. This pathogen (Mohanan and Sharma 1988) and other *Cylindrocladium spp*, including *C. scoparium* (Bertus 1976), *C. ilicicola* (Abraham *et al.* 1996), *C. crotalaria* (Peerally 1974a) and *C. theae*, (Peerally 1974b) cause foliar spots, blights and root disease in many acacia species, however significant disease impacts are usually limited to nurseries.

Symptoms and signs
Cylindrocladium species infect roots, collars, juvenile stem tissue and foliage of acacia seedlings. There are some reports of damage to acacias by *C. quinqueseptatum* after out-planting. Naturally regenerated seedlings, particularly if these develop under a forest

Fig. 39. Leaf and shoot blight of *Acacia auriculiform*is caused by *Cylindrocladium quinqueseptatum (C. reteaudii).* (J.K. Sharma)

Fig. 40 (right). Conidia of *C. quinqueseptatum.*

Fig. 41 (below). Conidiophore with conidia and vesicle.

canopy, can also become infected. On young phyllodes, grey water-soaked spots develop and eventually form extensive necrotic areas, usually along the margins and tips. Large numbers of white spores can be readily seen, by means of a hand lens, at lesion margins and more generally on necrotic tissue. Blighted shoots also bear masses of spores. Conditions of high humidity favour disease (Mohanan and Sharma 1988), for example, crowded nursery benches or plastic pots on the ground. Affected phyllodes turn reddish-brown (Fig. 39) and are prematurely shed.

Conidia of *Cylindrocladium* are typically cylindrical in shape with one or more cross walls. Figure 40 shows fruiting structures of *C. quinqueseptatu*, including the characteristic five celled macroconidia and vesicles at the tips of sterile hyphae (Fig. 41).

Hosts
The host ranges of the better-known and more virulent *Cylindrocladium* spp. e.g. *C. floridanum, C. quinqueseptatum* and *C. scoparium* are often wide (Crous and Wingfield 1994). Peerally (1991) pointed out that nursery diseases associated with *Cylindrocladium* infections may involve a complex of species. For *Acacia* spp. grown in plantations on a significant scale, there are records of *Cylindrocladium* infections of *A. auriculiformis, A. catechu, A. dealbata, A. mangium* and *A. mearnsii.*

Geographical distribution
Cylindrocladium spp. are widely distributed, with some species, e.g. *C. quinqueseptatum,* having a mainly tropical/sub tropical distribution, and others e.g. *C. scoparium,* being found worldwide. The taxonomy of this group has been subject to considerable attention over the past decade (Crous and Wingfield 1994) but the relationships between many species and their climatic and host requirements have yet to be established. The large numbers of *Cylindrocladium* species found in the humid tropics however, and common occurrence on some plantation crops, especially eucalypts, suggests their particular importance in these regions of the world.
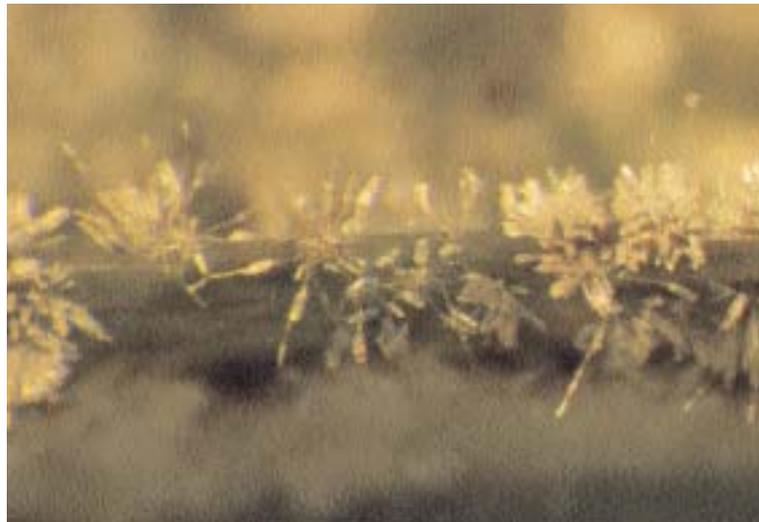

Fig. 42. Blighted shoot covered with conidiophores of *C. quinqueseptatum.*

Biology and transmission

Cylindrocladium spp. can infect almost any organ of seedlings and juvenile plants. More virulent species infect phyllodes of trees of any age after out-planting but there are no reports of significant damage to mature acacias. Several species produce chlamydo-spores. These are thick walled, often pigmented, propagules formed from hyphal cells, which allow the fungus to survive for extended periods in soil. Seedlings in contaminated nurseries are readily infected and lower whorls of juvenile trees in the field are exposed to spores splashed from the soil during heavy rain. The period from infection to sporulation can be as little as 3–4 days.

Coppice growth, juvenile in its physiology, sprouting from stumps after felling, or from trees recovering after insect defoliation, is highly susceptible to infection. Such infected trees maintain inoculum levels in the vicinity of plantations. Epidemic events are then driven by prolonged periods of high humidity and warm temperatures (Booth *et al.* 2000).

Detection

Infected leaves and shoots carry vast numbers of spores. These tend to aggregate together in clusters that are visible to the naked eye (Fig. 42). Isolation is very easy by transferring spore masses to agar with sterile needles, by plating infected leaf tissue or even by using leaves to bait the fungus from infected soil or leaf litter. The characteristic cylindrical, septate, conidia and protruding vesicles make identification to genus relatively simple. Further identification is more difficult due to the large numbers of species in this genus but useful keys are available (Crous and Wingfield 1994)

Treatment

None, infected germplasm should be destroyed.

Cercospora and *Pseudocercospora* foliar disease

Causal organisms
Two, as yet un-named foliar pathogens, in the genera *Cercospora* and *Pseudocercospora*.

Significance
A *Cercospora* sp. caused severe disease of seedlings of *Acacia mangium* and *A. auriculi-formis* raised in a nursery at Ingham in Queensland Australia in 1990 (Old *et al.* 1996). Seedlings from this nursery had already been planted out at several locations in northern Queensland and the pathogen became established at 9 of these sites. Despite annual visits to the region including sites where serious damage had occurred to *A. mangium*, the disease has not been found since 1991.

A *Pseudocercospora* sp. (Cannon *et al.* 1996; Old *et al.* 1997) has been found throughout a significant proportion of the natural range of *A. crassicarpa* in northern Queensland and on Melville Island, Northern Territory (Yuan 1996). This fungus also occurs on *A. flavescens.*

Experience in northern Queensland indicates that the *Cercospora* sp. associated with the Ingham outbreak is, potentially, a more serious pathogen than the *Pseudocercospora* sp. The impact of *Cercospora* sp. on seedlings in the nursery and young plantations of *A. mangium* was very damaging (Bruce Brown, personal communication); many trees died and the extensive infections of apical shoots caused multiple branching and severe loss of form. *Pseudocercospora* sp. in contrast causes phyllode necrosis and distortion of fine shoots in the crowns of saplings (Fig. 43), but no major damage has yet been recorded.

Fig. 43. Sapling of *Acacia crassicarpa* heavily infected by *Pseudocercospora* sp.

Neither of these fungi appear to have been have been recorded outside Australia, however several species in the genera *Cercospora* and *Pseudocercospora* have been described on *Acacia* from India. These include *P. acaciae* on leaves of *A. concinna* (Kamal 1980). Pongpanich recorded a *Cercospora* sp. as causing a pod rot of *A. auriculiformis* in Thailand (Pongpanich 1997). The relationship between these records and the Australian collections needs further study.

Symptoms and signs

Phyllodes bear spots, blotches and more extensive reddish-brown necrotic areas with scattered, or more densely arrayed sporulating structures (Figs 44 and 45). Conidia of

Fig. 44 (top). Detail of infected phyllodes showing distorted laminae and orange-brown lesions.
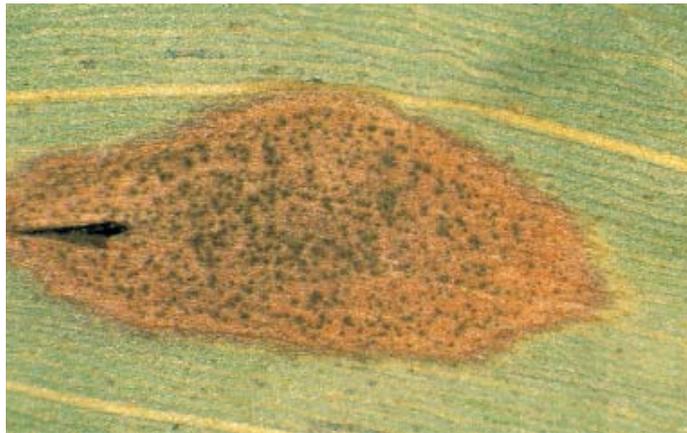
Fig. 45 (bottom). Necrotic lesion caused by *Pseudocercospora* sp. with fungus present on lesion surface.

Cercospora are 1–5 septate and non-pigmented, whereas *Pseudocercospora* spores are pigmented (Figs 46 and 47). Infected phyllodes often become distorted or crinkled (Fig. 44).

Hosts
Cercospora sp., *A. mangium, A. auriculiformis: Pseudocercospora* sp., *A. crassicarpa, A. flavescens: P. acaciae, A. concinna.*

Geographical distribution
Records are limited to northern Australia for the *Cercospora* and *Pseudocercospora* spp. described here although similar fungi have been collected on acacias in Thailand and India. The taxonomical affinities of these fungi makes it difficult to determine their distribution, especially as the *Cercospora* sp. causing severe damage in Queensland in 1990–91 has not been collected since that time.
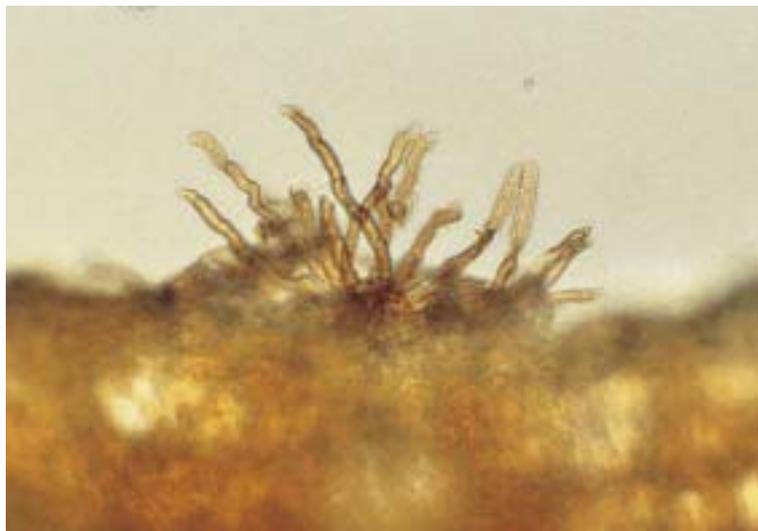

Fig. 46. Detail of sporulation with conidiophores and developing conidia.

Biology and transmission
The pathology of disease caused by *Cercospora* and *Pseudocercospora* on acacias has not been systematically studied. In common with some other *Cercospora* spp. these pathogens may be seed-borne. As the *Cercospora* outbreak in Queensland was first noticed in provenances of *A. mangium* originating in Papua New Guinea, it is possible that the fungus originated with the seed, warranting its inclusion in this manual. An intensive study of seed-lots in storage at the ATSC, Canberra was carried out but no *Cercospora* isolations were made from 35 bulk seed-lots imported from PNG (Old *et al*. 1996). The *Pseudocercospora* sp. found in plantations and native stands of *A. crassicarpa* in northern Australia, appears to be indigenous to the region.

Detection
Detection of these fungi and their identification to genus is relatively straightforward. Infected phyllodes show extensive, orange-brown necrotic areas bearing compact tufts of sporulating hyphae and cylindrical conidia, visible with a hand lens (Figs 45–47). Affected phyllodes, often high in the crowns or toward the apices of branches become distorted or crinkled. The fungi can be cultured on standard mycological media.

Fig. 47. Mature conidia of *Pseudocercospora* sp.

Camptomeris leaf spot

Causal organisms
Two species of *Camptomeris* have been recorded as pathogens of *Acacia mearnsii.* These are *Camptomeris albiziae* (Petch) Mason and *C. verruculosa* (Syd) Bessey.

Significance
Infection of pinnules by *C. albiziae* and *C. verruculosa* results in defoliation of *A. mearnsii* in South Africa, especially during late summer and autumn.

Symptoms and signs
Both species of *Camptomeris* cause small individual lesions on pinnules which become chlorotic (Fig. 48). Affected trees have sparse foliage due to premature shedding of older leaves as a result of infection.

Hosts
Only three plantation *Acacia* spp. have been noted as hosts of *C. albiziae* and *C. verruculosa.* They are *A. mearnsii* (Sherry 1971; Gibson 1975; Crous and Braun 1996; Wingfield and Roux 2000), *A. decurrens* (Bakshi 1976) and *A. saligna* (Crous and Braun 1996).

Geographical distribution
Camptomeris albiziae has only been reported from South Africa (Sherry 1971; Bakshi 1976; Wingfield and Roux 2000) and it is thought to be restricted to this country (Crous and Braun 1996). *Camptomeris verruculosa* on the other hand has been reported from Dominica, Kenya, South Africa and Sudan (Gibson 1975; Bakshi 1976).

Biology and transmission
There has been no systematic study of disease caused by *Camptomeris* spp. in acacia plantations.

Detection
The presence of erumpent, sporodochial fruiting bodies in pale/light brown spots on the under surface of leaves. The leaf spots are irregular in shape with a diameter of up to 3 mm.

Treatment
No treatment is recommended.

Fig. 48. Minute fruiting bodies of *Camptomeris albiziae*, on a pinnule of *Acacia mearnsii.*

Ceratocystis wilt

Causal organism
Ceratocystis albofundus De Beer, Wingfield and Morris and *C. fimbriata* Ell. & Halst.

Significance
Ceratocystis wilt pathogens can affect trees of all ages. *C. albofundus* results in a very rapid wilt and death of trees and is capable of killing one-year old *A. mearnsii* trees within 6 weeks after artificial inoculation (Roux *et al.* 1999). Apart from killing trees, infection, canker formation and gummosis makes stripping of the bark difficult. This is significant, as acacia bark is used in the tanning industry and de-barked logs are potential feedstock for pulp mills. In Brazil, *C. fimbriata* infection is reported to cause extensive stem canker, die-back, reduction in growth and death of *A. decurrens* (Ribeiro *et al.* 1988).

Symptoms and signs
Ceratocystis wilt of *Acacia* spp. is characterized by rapid wilt and die-back of infected trees. External symptoms of infection include the formation of stem cankers, exudation of gum, the formation of 'blister' type swellings on bark and discolouration of the bark around these lesions and cankers. Internal symptoms of *Ceratocystis* infection are very characteristic with lines or streaks of discoloured wood resulting from colonization of the vascular tissue (Fig. 49). Gum pockets are also commonly found in the wood of dying trees. Sometimes fruiting bodies of *Ceratocystis* (perithecia) can be found between the bark and the wood of dying trees.

Hosts
Ceratocystis albofundus has been recorded only from *A. mearnsii, A. decurrens* (Morris *et al.* 1994) and from native *Protea* spp. in South Africa (Morris *et al.* 1994). *C. fimbriata* has a wide host range including both woody and herbaceous hosts (Kile 1993). For plantation *Acacia* spp., *C. fimbriata* has been recorded only from *A. decurrens* (Ribeiro *et al.* 1988) and *A. mearnsii.*

Geographical distribution
Ceratocystis albofundus has been recorded only from Africa where it is known in South Africa and Uganda (Roux *et al.* 2001). It has been most commonly found in areas with temperate climates. *C. fimbriata*, however, has a worldwide distribution and has been recorded from many continents. On *Acacia* species it has, however, only been recorded from Brazil (Ribeiro *et al.* 1988) and South Africa (Roux 1998).

Biology and transmission
Ceratocystis species require wounds for infection. These wounds can be caused by mechanical damage, hail or insects. In South Africa, infection and disease caused by *C. albofundus* on *A. mearnsii* is especially evident after hail damage to trees.

Ceratocystis spp. can act in several ways to cause disease and death of trees. Some are canker and stain pathogens, while those infecting plantation *Acacia* spp. also cause vascular wilt. The pathogens may cause physical blockage of the vascular tissue through the stimulation of tyloses that occlude vessels, resulting in rapid wilt and death of trees. Many of the phloem, pith and primary xylem cells can also be plugged with hyphae. *Ceratocystis* spp. may also produce toxins, hydrolytic enzymes, and they can disrupt hormonal regulation (Kile 1993).

Many insects have been recorded as vectors of *Ceratocystis* spp. Species such as *C. fimbriata* and *C. albofundus* produce aromatic substances that are attractive to sap feeding insects such as those in the families Nitidulidae, Rhizophagidae, Staphylinidae and Drosophilidae. These insects visit fresh wounds on trees and thus transmit fungi to healthy trees (Kile 1993). Insects are also indirectly involved in the transmission of *Ceratocystis* spp. as their body surfaces are commonly coated with frass, contaminated with fungal spores, from galleries (Kile 1993).

Detection
Detection of *C. albofundus* and *C. fimbriata* in infected plant material can be difficult. Pieces of symptomatic wood (Fig. 49a and b) can be placed in moist chambers to induce the formation of the very characteristic fruiting bodies. Alternatively, symptomatic material can be baited using slices of carrot. This is done by wrapping symptomatic wood between two slices of carrot and incubating for 5–10 days in a moist environment.

Fig. 49 (a) and (b). Vascular streaking of *Acacia mearnsii,* typical of *Ceratocystis* infection.

If successful, fruiting bodies will appear on the carrot slices. Both pathogens are characterized by the formation of long necked perithecia, but *C. albofundus* can be distinguished from *C. fimbriata* by the fact that its perithecial bases are light (cream) coloured (Fig. 50) compared to the black bases of *C. fimbriata* (Fig. 51).

Treatment
No eradicative treatment is available. Impact in plantations can be reduced through selection of disease tolerant material and avoiding wounding to trees.

Fig. 50 (top). Perithecia of *C. albofundus*, only the perithecial necks are pigmented.

Fig. 51 (below). Darkly pigmented perithecium of *Ceratocystis fimbriata.*

Fusarium canker and wilt

Causal organisms
Numerous *Fusarium* species have been associated with diseases of plantation *Acacia* spp. These include *F. accuminatum* Ell. & Ev. Sensu Gordon, *F. graminearum* Schwabe (Aoki and O'Donnell 1999 = *F. graminearum* group 2 sensu Francis and Burgess 1977), *F. solani* (Mart.) Saac, *F. oxysporum* Schlecht, *F. subglutinans* (Wollenw. & Reinking) Nelson, Toussoun & Marasas and *F. proliferatum* (Matshushima) Nirenberg.

Significance
In nurseries, *Fusarium* spp. can cause rapid and extensive damping-off of young seedlings. No serious plantation diseases have been attributed to infection by *Fusarium* spp., but *F. solani* has been associated with a wilt disease in India (Lenné 1992). *F. graminearum* has recently been reported to cause stem cankers and die-back of *A. mearnsii* branches in South Africa (Roux *et al.* 2001).

Symptoms and signs
The most common symptoms associated with *Fusarium* infection in nurseries are: the occurrence of post emergence damping off, leaf drop and lesions / cankers on the stems of young trees (Bakshi 1976; Gibson 1975; Old *et al.* 2000). Under field conditions, on older trees, infection by *Fusarium* spp. has been associated with stem and branch cankers (Roux *et al.* 1997) as well as wilt of trees (Lenné 1992). Infection by *F. graminearum* may also lead to branch die-back (Roux *et al.* 1997, 2001). Species such as *F. accuminatum*, *F. subglutinans* and *F. proliferatum* have also been associated with branch and stem cankers (Roux *et al.* 1997). Fruiting bodies and mycelium of both the asexual and sexual states of these *Fusarium* spp. are often found sporulating on the surface of moist cankers and lesions.

Hosts
Fusarium species have wide host ranges especially on herbaceous plants. Woody hosts include *A. catechu* (Lenné 1992), *A. dealbata* (Gibson 1975), *A. decurrens* (Gibson 1975; Bakshi 1976), *A. mangium* (Zakaria 1990), *A. koa* (Zakaria 1990), *A. auriculiformis, A. holosericea* and *A. mangium* (Old *et al.* 2000).

Geographical distribution
Fusarium species associated with diseases of plantation-grown *Acacia* spp. have been recorded in South Africa, India, Malaysia, Indonesia and Japan (Gibson 1975; Lenné 1992; Zakaria 1990; Old *et al.* 2000).

Biology and transmission
Fusarium spp. display many different types of infection and transmission strategies. Many are generally stress-related pathogens and require wounds for infection.

Increased levels of nitrogen fertilization in nurseries have been shown to increase the incidence and severity of *Fusarium* infection.

In nurseries, water, growth media and contaminated seed have been shown to be sources of *Fusarium* infection. Many *Fusarium* spp. also have close associations with insects as vectors and in providing wounds for infection. Other species are adapted for wind dispersal (Nelson *et al.* 1981).

Detection
Under favourable conditions, *Fusarium* spp. sporulate on fresh cankers and wounds, making identification to the genus level possible directly from plant material. Diseased material can also be placed in moist chambers for isolation. Isolations by transfer of diseased material directly onto standard mycological media (Nelson *et al.* 1983) are also highly successful. Identification of *Fusarium* spp. to the species level requires specialized media. For some species rapid molecular techniques are available to aid in diagnosis.
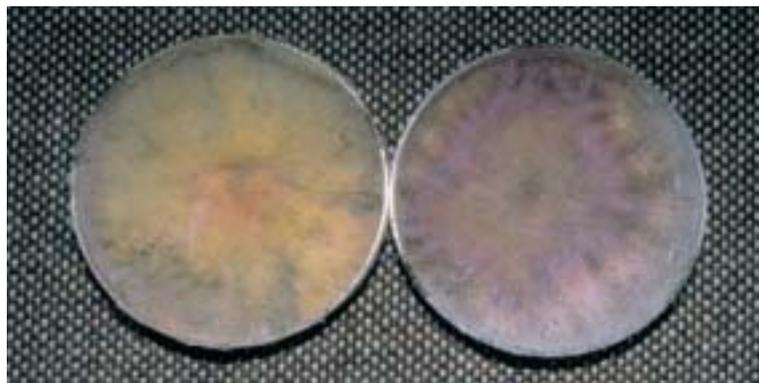
Fig. 52. Cultures of *Fusarium graminearum* associated with cankers and die-back of *Acacia mearnsii.*

Treatment
No treatment is available for *Fusarium* infection on trees in plantations. It is, however, common practice to treat nursery infections of *Fusarium* with fungicides. Both contact and systemic fungicides are useful in this case. Strict nursery management is essential for preventing losses caused by *Fusarium* spp. It is crucial to ensure that all water, growth media and containers are free of any inoculum and that only pathogen free seed is used for planting.

Fig. 53. Stem canker of *A. mearnsii* resulting from inoculation with *F. graminearum.*

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APPENDIX I.
PATHOGENIC FUNGI ASSOCIATED WITH ACACIA SEEDS

Fungus	Host	Country	References
Botryodiplodia theobromae	*A. confusa*	Philippines	Agmata 1979
(syn. *Lasiodiplodia theobromae*)	*A. auriculiformis*	Thailand	Chalermpongse *et al.* 1984
Botryodiplodia sp.	*A. auriculiformis*	Australia	Yuan *et al.* 1997 (unpubl.)
Colletotrichum gloeosporioides	*A. auriculiformis*	India	Mohanan and Sharma 1988
Colletotrichum sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
Curvularia brachyspora	*A. auriculiformis*	Thailand	Chalermpongse *et al.* 1984
Curvularia eragrostidis	*A. auriculiformis*	Australia	Yuan *et al.* 1997 (unpubl.)
Curvularia lunata	*A. auriculiformis* *A. catechu* *A. confusa* *A. crassicarpa*	Australia India Philippines Australia	Yuan *et al.* 1990 Vijayan 1988 Agmata 1979 Yuan *et al.* 1997 (unpubl.)
Curvularia pallescens	*A. auriculiformis*	Philippines	Mittal *et al.* 1990
Curvularia senegalensis	*A. auriculiformis*	Australia	Yuan *et al.* 1990
Curvularia sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
Cylindrocladium sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
Diplodia sp.	*A. crassicarpa*	Australia	Yuan *et al.* 1997 (unpubl)

(cont.)

Fungus	Host	Country	References
Drechslera spp.	*A. auriculiformis*	Thailand	Pongpanich 1997
	A. crassicarpa	Australia	Yuan *et al.* 1997 (unpubl.)
Fusarium equiseti	*A. catechu*	India	Vijayan 1988
Fusarium moniliforme	*A. raddiana*	Israel	Mittal *et al.* 1990
Fusarium oxysporum	*A. catechu*	India	Vijayan 1988
Fusarium semitectum	*A. auriculiformis*	Philippines	Mittal *et al.* 1990
	A. auriculiformis	India	Mohanan and Sharma 1988
	A. modesta	India	Mittal *et al.* 1990
Fusarium solani	*A. catechu*	India	Vijayan 1988
	A. holosericea	Australia	Yuan *et al.* 1990
Fusarium sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
	A. auriculiformis	Australia	Yuan *et al.* 1990
	A. auriculiformis	Australia	Yuan *et al.* 1997 (unpubl.)
Hansfordia sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
Helminthosporium sp.	*A. mearnsii*	Australia	Yuan *et al.* 1990
Lasiodiplodia sp.	*A. auriculiformis*	Thailand	Pongpanich 1997
Pestalotia sp.	*A. mearnsii*	China	Liu 1988
Pestalotiopsis disseminata	*A. auriculiformis*	Australia	Yuan *et al.* 1997 (unpubl.)
Pestalotiopsis neglecta	*A. auriculiformis*	Australia	Yuan *et al.* 1997 (unpubl.)

(cont.)

Fungus	Host	Country	References
Pestalotiopsis phoenicis	*A. auriculiformis*	Australia	Yuan *et al.* 1997 (unpubl.)
Pestalotiopsis sp.	*A. auriculiformis*	Australia	Yuan *et al.* 1990
Phoma sp.	*A. auriculiformis*	India	Mathur 1974
	A. auriculiformis	India	Mohanan and Sharma 1988
	A. confusa	Philippines	Agmata 1979
	A. modesta	India	Mittal *et al.* 1990
	A. raddiana	Israel	Mittal *et al.* 1990
	Acacia sp.	Egypt	Mittal *et al.* 1990
Phomopsis sp.	*A. auriculiformis*	Thailand	Pongpanich 1997

Source: Old and Yuan 2001, *Tropical Tree Seed Manual*, USDA Forest Service.

APPENDIX II.
COMMENTS ON TECHNICAL GUIDELINES FOR THE SAFE MOVEMENT OF *ACACIA* GERMPLASM

please send to:

Germplasm Health Scientist   or Forest Resources Development Service
IPGRI-Americas   FAO
AA 6713, Cali, Colombia   Via delle Terme di Caracalla
Fax: 57-2-4450073   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 _____
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Date   Signature

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