Diseases of *Populus* in British Columbia:

A Diagnostic Manual

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Contents

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Brenda Callan 1a, 1b, 1c, 1d, 1e, 1f, 1k,1l, 1n, 1o, 1p, 1q, 2a, 2b, 2i, 2k, 2n, 3a, 3b, 3c, 3d, 4a, 4b, 4c, 4d, 4f, 5b, 5d, 5g, 6a, 6b, 6c, 6d, 6e, 7b, 7c, 7d, 7e, 8a, 8b, 8c, 8d, 9a, 9b, 10c, 11j, 12b, 12c, 12d, 13b, 15a, 15b, 15c, 15d, 15e, 15g, 16d, 16e, 16f, 16i, 17a, 17b, 17c, 17f, 17g, 17j, 17k, 17l, 17m, 17n, 17o, 18a, 19a, 19b, 19c, 19d, 20a, 20b, 20d, 21d, 21e, 22a, 22c, 23a, 23b, 23c, 23d, 24a, 24b, 24c, 24d, 24e, 24f, 25b, 26a, 26b, 26c, 26d, 26g, 27a, 27c, 27d, 27e, 28a, 28b, 28c, 28f, 28g

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Cover photographs by Brenda Callan. Clockwise from left: nine-year-old hybrid poplar plantation in the Fraser Valley; autumn colors of cottonwood and aspen along the Ft. Nelson River; hybrid poplar nursery in the Fraser Valley.

How to Use this Book

This diagnostic manual is designed to help the reader with both field and laboratory identification of common poplar diseases in British Columbia, and to provide regionally specific disease impact and distribution information, where it is known.

The reader is first referred to the "Field and Laboratory Techniques" section where proper field collection techniques are described.

Once the field observations and collections have been completed, other sections in the book aid in the identification process:

- dichotomous keys, based primarily on symptoms and signs visible in the field or by examination using a hand lens
- detailed descriptions, including microscopic features, accompanied by color photographs
- host-fungus index, which may be used to confirm whether or not the pathogen has been previously reported on a given host
- disease distribution maps, which are especially useful for geographically limited species such as *Armillaria ostoyae*. Not every pathogen description is accompanied by a disease distribution map because certain pathogens are poorly documented on their poplar hosts in British Columbia.

The Poplar Resource in British Columbia

The British Columbia (B.C.) forest industry is currently interested in the use of native aspen, balsam poplar and cottonwood (genus *Populus*) for wood products such as oriented strand board, veneer, chopsticks, and pulp (Peterson and Peterson 1992). In addition to the harvest and tending of subsequent natural regeneration of native poplar species, forest companies are exploring the intensive cultivation of fast-growing hybrid poplars for wood fibre (McLennan and Mamias 1992). With the growing interest in this valuable hardwood resource comes the need for pathology research and disease diagnostic tools specific to our region (Newcombe 1996). This manual expands upon basic mycology information provided in previous regional manuals (Funk 1981, 1985). It is also intended to complement similar manuals designed for other regions (DeByle and Winokur 1985; Ostry et al. 1988).

Until the 1980s, aspen was mostly regarded as a weed tree by the B.C. forest industry. Coniferous tree species such as spruce were considered more valuable, and aspen was viewed with disfavor as competing with spruce and interfering with its harvest. Today, with reduced allowable annual cuts, aspen is now regarded as a resource in its own right, is managed for harvest, and is used in such products as oriented strand board, pulp, and chopsticks. For example, in 1990, the B.C. Forest Service granted rights to harvest 1.4 million m^3 of aspen per year in the Peace River District (Lance et al. 1996).

Black cottonwood and balsam poplar are the fastest growing native trees in B.C. (McLennan and Mamias 1992). As early as the 1950s, southern coastal forest companies were interested in establishing hybrid poplar plantations and harvesting native cottonwood, primarily for making plywood veneer (Smith 1966). In the 1980s a second wave of interest in hybrid poplar cultivation in south coastal B.C. emphasized short-rotation, intensively managed plantations for the production of pulp. Many of the hybrids planted in trials and commercial plantations were produced from research programs at the University of Washington and Washington State University (Stettler et al. 1996). In these breeding programs, hybrids between native cottonwood, *P. trichocarpa* and other nonnative species, particularly in sections *Aigeiros* and *Tacamahaca* (see the following section on taxonomy) have shown amazing levels of hybrid vigor, or heterosis.

Hybrids often show growth form and productivity superior to that of their nonhybridized parents (Stettler et al. 1996). Hybridization also permits the introduction of disease resistance characters which may be present in one parent but not the other. However, to realize their high growth potential, hybrids must be intensively cultivated in an agricultural regime, with extensive site preparation, fertilization, and strict control of competing vegetation for the first 3 years until canopy closure is achieved. (McLennan and Mamias 1992).

Cottonwood and hybrid poplar pulp features high opacity, good bulk, and printability; it improves the quality of newsprint when combined in low percentages with softwood kraft pulp. Poplar pulp also provides superior fibre for the manufacture of value-added grades of paper, such as computer paper, book paper, and offset printing paper. As of 1992, companies using cottonwood as a major source of raw material in B.C. consumed about 500 000 to 600 000 m^3 per year (McLennan and Mamias 1992).

Extensive monographs on the biology and ecology of *Populus* in B.C. and western Canada have been recently compiled by several authors (Comeau et al. 1996; Peterson et al. 1996).

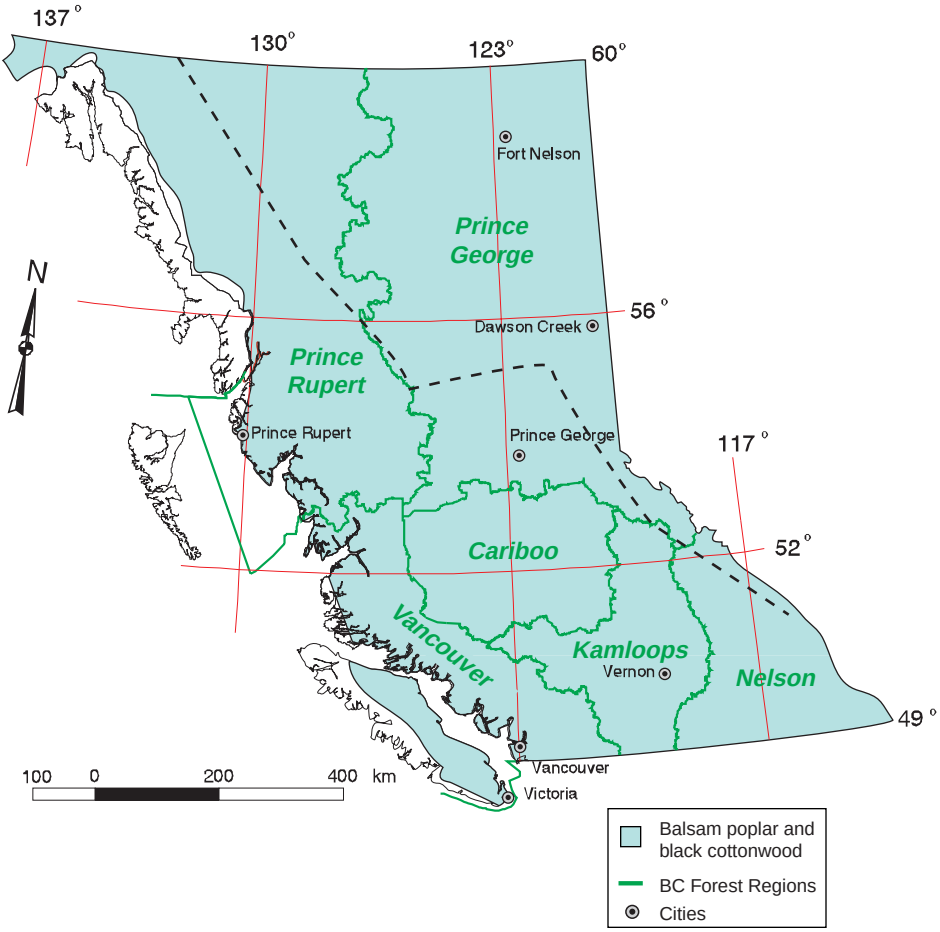
Taxonomy and Distribution

The morphologically and ecologically distinct groups of poplar species are usually separated into six different sections. The major barriers to hybridization occur between sections.

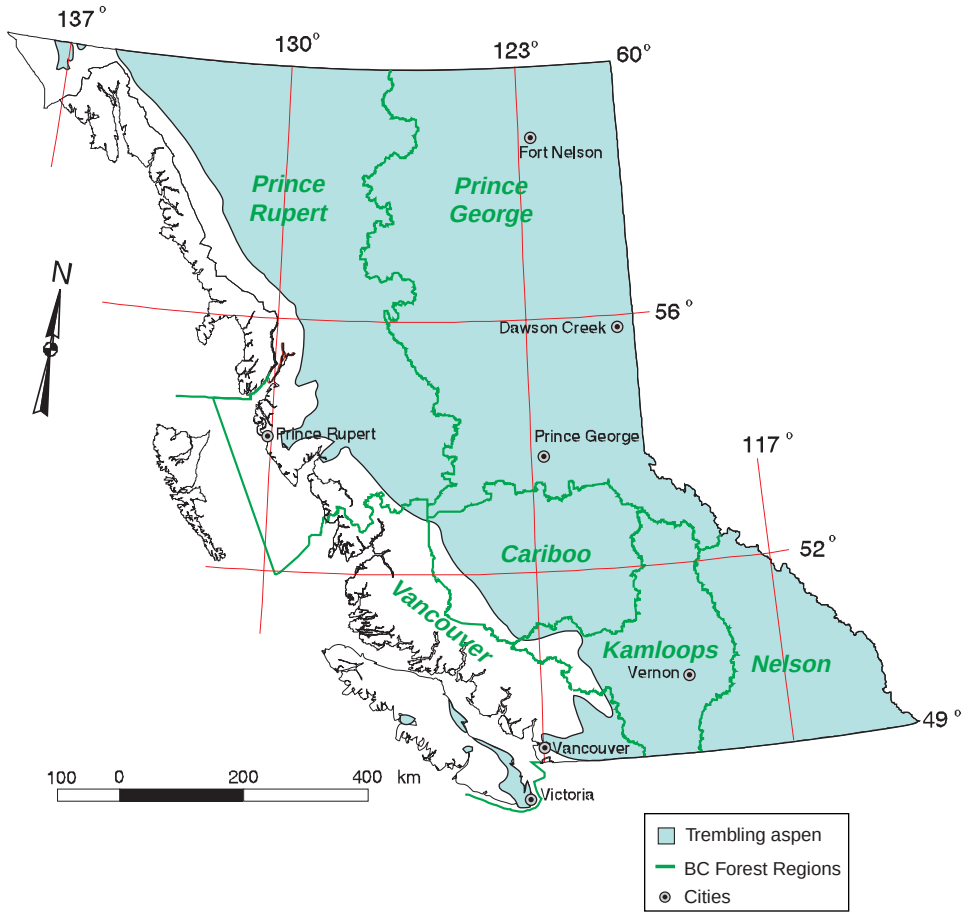
The six sections in the genus *Populus* are listed. The first three sections do not include species relevant to this manual, which follows the taxonomic system accepted and described by Eckenwalder (1996). This system provisionally accepts 29 species in the genus worldwide. Species discussed in this manual are listed after the appropriate section names; those species in **bold** are native to B.C. The single letter in parentheses is the abbreviation used to identify hybrid crosses.

1. Section *Abaso* Eckenwalder
2. Section *Turanga* Bunge
3. Section *Leucoides* Spach
4. Section *Aigeiros* Duby includes:
   - *P. deltoides* Marshall (D): Eastern cottonwood is native to eastern North America.
   - *P. nigra* L. (N): Black poplar is native to Eurasia. *Populus nigra* var. *italica*, Lombardy poplar, is a male tree used for windbreaks in Europe and North America.

   Both of these species have been hybridized with black cottonwood for commercial production in the Pacific Northwest.
5. Section *Tacamahaca* Spach includes:
   - *P. balsamifera* L.: Balsam poplar is native to North America, and occurs in northern and interior regions of B.C. but readily hybridizes with *P. trichocarpa* where their distributions overlap (see distribution map on page 3).
   - *P. suaveolens* Fischer, *sensu lato* (M). Japanese poplar is native to Eurasia. In this manual this species is referred to as *P. maximowiczii* which is the name traditionally used in breeding literature (Zsuffa 1975). This species has been hybridized with black cottonwood for commercial production in the Pacific Northwest.
   - *P. trichocarpa* Torr. & Gray (T): Black cottonwood is native to North America, and occurs in western B.C. (see distribution map on page 3) but readily hybridizes with *P. balsamifera* where their distributions overlap.
   - *P. angustifolia* James: Narrow-leaf cottonwood.
6. Section *Populus* includes:
   - *P. alba* L.: White poplar is native to Eurasia.
   - *P. grandidentata* Mich.: Large-toothed aspen is native to eastern North America.
   - *P. sieboldii* Miquel.: Japanese aspen is native to eastern Eurasia.
   - *P. tremula* L.: European aspen is native to Eurasia.
   - *P. tremuloides* Mich.: Trembling (quaking) aspen is native to North America, and occurs throughout B.C. (see distribution map on page 4). It is most common in the central and northern interior of the province, but is sparsely distributed on the west coast. *Populus tremuloides* var. *vancouverianae* (Trel.) Sarg. is confined to Vancouver Island, while the variety *aurea* (Tid.) Dan. occurs elsewhere in the province (Krajina et al. 1982).



Distribution of balsam poplar and black cottonwood (*Populus balsamifera* L. and *Populus trichocarpa* Torr. & Gray). Dashed line represents the approximate boundary between the distributions: balsam poplar to the north-east, black cottonwood to the south-west. The italicized names indicate the six provincial forest regions. Map adapted from Krajina et al. 1982.



Distribution of trembling aspen (*Populus tremuloides* Mich.). The italicized names indicate the six provincial forest regions. Map adapted from Krajina et al. 1982.

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Field Collection of Tree Diseases

A diagnosis in the field is not always possible, especially if the pathogen is immature, or if affected tissue requires incubation, culturing, or microscopic examination to determine the causal agents. If an expert has agreed to assist with a laboratory diagnosis, a number of steps may be taken in the field to send the best material thus ensuring proper identification. Collector name and address, date of collection, the host species, symptoms, and geographic location should always be included with each specimen. Many agencies will provide a collection data sheet for this purpose.

Use of the proper collection techniques outlined below will increase the likelihood of a correct diagnosis.

Foliar diseases

- Cut one or more lengths of branch or stem, about 20 cm long, with diseased foliage attached.
- Include a second cutting with healthy foliage, for comparison.
- Include flowers if species is unknown.
- Press the freshly cut foliage between folded sheets of dry newspaper (not plastic).
- Package specimen flat in a paper envelope or in a cardboard box.
- If collection is made in spring or early summer, fungi developing on current-year growth may be immature. Search for overwintered foliage in ground litter or branch crotches, and include a separate collection, either pressed or wrapped in a paper bag, with the fresh sample.
- Do not moisten or seal the specimen in plastic wrapping, as this encourages contaminants to grow. Preferably let the material dry during storage or transit.

Stem or branch cankers (< 10 cm in diameter)

- Sample should include both living and dead host tissue, if possible, because the pathogen is most active in dying tissue (i.e., be sure to cut stem or branch a few centimeters below the visible edge of the canker).
- Saw or clip a 20-cm-long section from cankered area of branch or stem.
- Pack sample in newspaper (not plastic), or roll it inside a large paper bag.

Stem or branch cankers (> 10 cm in diameter)

- If tree is to be felled, or if it is possible to remove a large branch, cut a 10- to 20-cm-long section of canker, preferably at the lower edge of the canker where the likelihood of living tissue is greatest.
- If tree cannot be felled, use a hammer and chisel, knife, or axe to cut out a 10×10 cm section of bark from the edge of the canker.
- If fruiting bodies are visible on other parts of the canker, remove a similar-sized portion from this area, too.
- Wrap the sample in newspaper (not plastic), and pack it in a padded paper envelope or cardboard box.

Wood decay samples

- Look for fruiting bodies near the decay column.
- Break off or cut out the conk, wrap it in newspaper, and, if it is not fragile, include in package with decay sample. If mushrooms are observed, collect and process as described in following sections.
- To avoid damage during transit, do not package large pieces of wood with fragile fruiting bodies.

- Document the type of wood decay (e.g., white rot, laminated decay, white pocket rot).
- To collect a sample of wood decay, cut into wood near the conk or point of breakage using an axe or chainsaw. Ideally, the wood sample should contain symptoms of advanced decay, incipient decay, and sound wood. These conditions are found near the edge of the decay column. Wood samples should be at least 15 × 15 × 15 cm, large enough to split with an axe.
- Wrap the samples in newspaper (not plastic), and pack them in a cardboard container.

Standing dead or dying trees with suspected root disease

Note: Long-dead, standing trees are generally colonized with saprophytic decay fungi. These fungi often produce mycelial fans or decay columns which extend several meters up the trunk of a dead tree, and are sometimes confused with pathogenic fungi. Carefully excavate down the butt and along roots of the tree until signs of pathogen activity, such as exudates, decay, or stunted, callused roots, are encountered. Samples should be taken from these areas.

- Samples should be taken from a dying or recently dead tree in the stand. Such trees are often found in a root disease center, between the inner most old dead trees, and symptomless trees at the outer edge.
- Samples should be taken from ground-level areas of the butt of the tree where signs (mushrooms, conks) or symptoms (heavy sap exudation, cracked bark at butt of tree) are visible. Use an axe, or chainsaw, and proper safety techniques, either falling the tree or removing from a standing tree a chunk of the outer part of the butt, 20 × 20 × 10 cm. It is important to leave the bark intact on the sample.
- If no root disease symptoms or signs are apparent on the butt of the tree, and the above-ground cambium reveals no evidence of root diseases (e.g., stains, decay, or mycelial fans), excavate the roots at the base of the tree, looking for oozing, stains, decay, or mycelial fans, and obtain a sample as previously described, if such signs are found. If the root is less than 10 cm in diameter, cut out a 20-cm-long, whole section of root, with bark intact.
- Wrap the sample in newspaper (not plastic), and pack it in a cardboard container.

Blowdown with decay

Note: Fungi fruiting on long-dead, fallen trees may not be the same ones responsible for tree failure.

- Identify the host species as accurately as possible.
- If the tree is too old to identify, note the other tree species in the vicinity.
- If the stem has failed, cut into wood near conk or point of breakage, but avoid heavily weathered or soiled areas. Sample the wood as described earlier.
- If roots have failed, check for root decay and sample from decayed roots, either those remaining in the ground or larger roots in the exposed root plate.
- Wrap the sample in newspaper (not plastic), and pack it in a cardboard container.

Wood decay mushrooms

- Follow instructions for collecting a wood decay sample.
- If possible, collect several mushrooms, especially if they are at different levels of maturity.
- *Temporary in-field packaging:* Pack the fungus in a paper bag (never plastic), or wrap it in wax paper. Roll the wax paper in a cylinder around the mushroom, then twist each end as if it were a candy wrapper.
- Carry mushrooms in a rigid container so that they do not get squashed.

- Process the specimens the same day they are collected, using the following guidelines:
  1. Obtain a spore print. Mushroom identifications depend on spore color. A spore print is very helpful but may only be obtained while the specimen is very fresh. To make a spore print, cut the stalk off a mature mushroom and place the cap gill-side down on a piece of white paper. If you suspect the spores might be white (check the stalk or the ground underneath), use colored paper. Cover the cap with a drinking glass or bowl if it is a very dry day. Consider making the spore print outside if the mushroom was collected on a cold day, as it might stop sporulating when warmed up to room temperature inside. Spore prints appear in several hours or overnight. Protect the spore print between cardboard before shipping.
  2. Air-dry, or heat-dry the sample. Most unpreserved fungi turn to mush during the few days it takes to send a sample by mail to an expert. Specimens should be dried before shipping unless special courier arrangements have been made. Small, fragile specimens under dry conditions may be air-dried in opened paper bags or wax paper in a warm, dry room for a day or two. Large fungi should be quickly dried using a portable dehydrator (such as those sold for drying food), otherwise they will decay or become infested with maggots. As a last resort, with careful and constant monitoring, a conventional oven may be used if turned to the lowest setting with specimens on the highest possible rack with the door open. Alternatively, use the warming drawer below the oven, left partially open while the oven is in use. Ideally, drying temperatures should be around 50°C.
- Pack dried specimens in rigid containers, or in zip-locked plastic bags packed in a box filled with foam chips. *Dried specimens are very fragile*.

Common Mounting Solutions Used for Microscopic Examination

Unless otherwise specified, assume that the spores in this manual were measured from sections or scrapings mounted in a drop of tap water on a microscope slide. The microscopic features of some fungi, such as *Marssonina* spp., are described here and elsewhere in the literature from spores mounted in lactophenol or lactoglycerol (the latter is a less toxic substitute preferred by the author).

To prepare lactoglycerol, dissolve a 10.0 g solution of lactic acid (specific gravity 1.21) in a solution of 10.0 ml of distilled water and 20.0 ml of glycerol. Add 10.0 g of pure phenol crystals to make lactophenol. Cotton blue (a 1% solution filtered after addition to remove undissolved crystals) or fuchsin stains may be added to lactoglycerol to differentiate fungal tissue from plant substrate. This mounting agent clears tissues, and is useful for making semipermanent slides if cover slips are sealed with nail varnish.

Diagnostic Reagents

Some fungal taxa undergo characteristic color changes when tested with standard mycological reagents. These reagents are usually added to tissue mounted in water on a microscope slide, and color changes are then observed microscopically. The three following reagents are commonly used.

Melzer's reagent

Dissolve 1 g potassium iodide, 0.3 g iodine, and 20.0 g of chloral hydrate in 20 ml of distilled water. When added to fungal tissue, a color change to blue is called an amyloid reaction. A color change to red or brown is called a dextrinoid reaction.

Potassium hydroxide

A 10% solution of potassium hydroxide dissolved in distilled water is used for observing color changes in ascomycetes. A 5% solution is traditionally used for softening tissue and observing color changes in basidiomycetes.

Sulfovanillin

Dissolve 1 g vanillin and 8 ml 70% sulfuric acid in 4 ml of distilled water (short shelf life and strong smell). Alternatively, dissolve 2–3 vanillin crystals in one drop of sulfuric acid on a microscope slide for individual mounts. This reagent is used to stain structures (cystidia, which turn black) in some wood decay fungi such as *Peniophora*.

All reagents and mounting solutions should be used following appropriate safety and disposal procedures.

The term “abiotic disease” refers here to any disorder directly caused by non-living stress factors. Abiotic diseases are usually associated with meteorological extremes affecting temperature and moisture levels, but human-caused problems such as air pollution or improperly applied chemicals also cause disease.

There are a few field indicators which, if observed, are useful for separating abiotic from biotic diseases when signs of the latter are not evident. As a rule, symptoms of non-infectious diseases are uniform on all sides of the tree exposed to the damaging agent. If the damaging agent comes from a point source (i.e. smoke stack or chemical dump), disease symptoms will increase in intensity closer to the source of emission. Conversely, symptoms of infectious diseases are variable and more random, scattered among healthy growth of the same age. In addition, abiotic damage tends to affect a wide variety of adjacent unrelated plants, while biotic disease agents are usually host-specific.

Chronic damage due to urban pollutants such as ozone and acid rain is rare in B.C. but well described in other regions (Jacobson and Hill 1970; Malhotra and Blauel 1980; Skelly et al. 1985). Specific examples of abiotic damage are described and illustrated in the following pages (Fig. 1).

Fig. 1. Abiotic and virus-like damage.

Figs. 1a, b. Virus-like foliar symptoms; variegation in cottonwood (Fig. 1a) and ring-spots (chlorotic haloes) on T×D hybrids (Fig. 1b). Although the symptoms are virus-like in nature, the actual cause is unknown. Phosphorous deficiency is also known to cause variegation in some plants (Malhotra and Blauel 1980). Affected trees are often stunted and slow-growing, which is another sign of a virus infection. Viruses on poplars have not been well studied.

Fig. 1c. Burl on trembling aspen. Burls are caused by abnormal epicormic bud proliferation, resulting in hypertrophied wood with a swirling, irregular grain. They are often produced at or near the base of the tree, and some at least develop in response to wounding (Sinclair et al. 1987). Wood inside burls is usually sound and highly figured. No pathogens are known to be specific causal agents of burls on poplars.

Fig. 1d. Abnormal callus development in a wounded trembling aspen. Such anomalies are often characteristic of individual aspen clones. The cause is unknown.

Figs. 1e, f. Frost rib on trembling aspen. Frost ribs develop after the trunk is cracked while the tree is dormant. The crack forms in response to a sudden severe drop in temperature, when the outer wood becomes far colder and shrinks more rapidly than the warmer inner wood. Cracks usually originate at the butt of the tree and may extend several meters up the trunk. The ridges of callus, which are produced in an attempt to heal the tree, may become pronounced if the crack is repeatedly opened by cold or strains induced by wind (Boyce 1961).

Fig. 1g. Frost damaged black cottonwood. The heaviest damage occurs when temperatures drop below freezing in spring at the time when newly flushed foliage is still soft and tender. Fully expanded, hardened foliage is more resistant to frost damage. Blackened tips of shoots and leaves may be confused with such diseases as Venturia blight (described elsewhere). If entire shoots are frost-damaged, regrowth later in the spring may be patchy. Trees which are frost-damaged for several consecutive springs may suffer from poor crown form and increment loss (Hiratsuka et al. 1995). An early autumn frost may injure bark that has not yet hardened for the winter.

Figs. 1h, i. Torn bark and shredded foliage on T×D hybrid poplars in Oregon state, following a severe hailstorm. Unlike canker diseases, hail damage on stems only occurs on the same side as the prevailing storm winds. Cankers caused by living organisms may occur on any side of the tree.

Fig. 1j. Blowdown in a T×D hybrid poplar plantation, caused by water-saturated soils and heavy winds. Fallen trees often lie parallel to the direction of the wind.

Fig. 1k. Wire girdling of a trembling aspen. Swelling of tissues above the wire is caused by a concentration of photosynthates where translocation is reduced or interrupted.

Fig. 1l. Scar from a mechanically torn branch on a T×D hybrid poplar.

Fig. 1m. Axe wounds on trembling aspen are commonly encountered in and around campsites.

Figs. 1n, o. Herbicide (simazine) spray drift damage on T×D hybrid poplars. Typical symptoms of herbicide damage include twisting and curling of leaves, necrotic or chlorotic spots in a "spray" pattern on foliage, and overall foliar chlorosis or necrosis. Severely damaged plants often have thin, twisted and curled shoots devoid of leaves. Similar patterns of deformities are usually present on other nearby plant species. (Hiratsuka et al. 1995).

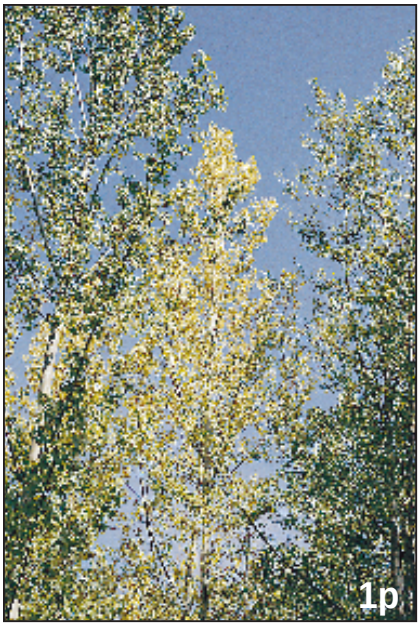

Fig. 1p. Fertilizer burn on T×D hybrid poplar. Damage caused by excessive application of fertilizer may result in chlorosis or scorched foliage. Spray drift may cause necrotic flecking.

Fig. 1q. Foliar symptoms of unknown nutrient deficiency on hybrid (T×D) poplar. Nutrient imbalances are often exacerbated by low or high soil pH, which limits the availability of certain minerals, such as iron or phosphorous, to the tree even if they are present in the soil, while making toxic elements such as aluminum more available (Skelly et al. 1985). Unless a foliar and soil analysis is performed, nutrient deficiencies are hard to detect because their symptoms are non-specific and resemble drought, frost, or even herbicide damage. Nitrogen, and phosphorous deficiencies cause both stunting and chlorosis, while low phosphorous tends to retard and stunt foliar development. Symptoms are often a combination of several deficiencies (Malhotra and Blauel 1980).

Fig. 1r. Salt spray (oceanside) damage of cottonwood and adjacent tree species. Salt damage is common on the coasts, on some occasions further than 10 km inland (after storms), and on roadsides. Roadside damage is worst on curves where more salt is applied. Uptake occurs from wind-driven sprays, or in the case of roadside damage, from root absorption of salts accumulated in the soil. Foliage shows marginal chlorosis and necrosis, and then drops prematurely (Skelly et al. 1985).

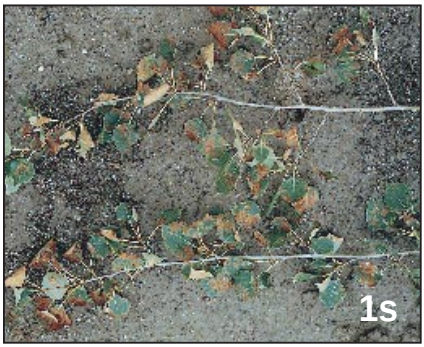

Fig. 1s. Smelter (SO_2) emission damage on trembling aspen occurs when the trees are exposed to fumes produced by smelting ores, manufacturing steel or refining petroleum. Damage is restricted to areas immediately downwind from point sources, unless the industrial source has tall smokestacks, in which case fume damage may occur up to 16 km downwind. Acute damage results in yellow to brown interveinal necrosis, with symptoms more pronounced at the periphery of the leaf. As the injured tissues become more weathered, they tend to darken. Recently matured leaves are the most sensitive to sulfur dioxide (Skelly et al. 1985).

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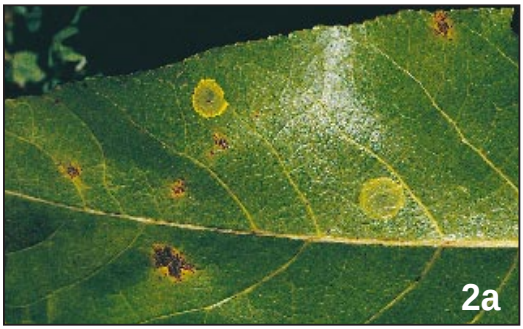
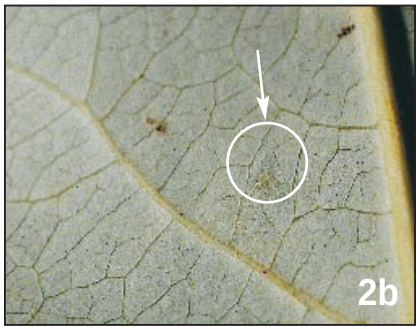
Arthropod Damage Often Confused with Poplar Diseases

Figure 2 illustrates symptoms of a number of common insect and other arthropod pests of poplars. These pests have been chosen because the damage they cause may at first be confused with canker diseases or foliar blights. A brief description of each figure, the causal agent, and citation of pertinent references follows.

Fig. 2. Common arthropod pests of poplars.





Figs. 2a, b. Cecidomyid galls (*Harmandia* sp.) form on leaves of cottonwood, balsam, and hybrid poplars (Whitney and Baranyay 1968). Developing galls (Fig. 2a) appear as chlorotic spots 5 mm wide on the cottonwood leaf. Spots appear near the end of June and turn brown 2 weeks later. A small, hyaline cecidomyid larva (Fig. 2b) is feeding on the underside of the circular lesion seen in Fig. 2a.

Figs. 2c, d. *Saperda calcarata* Say (Coleoptera, Cerambycidae) feeding damage on aspen, which is the primary host in B.C. Other poplar species and *Salix* are also attacked by this insect, whose larvae are legless, cream-colored, and 40 mm long. The adult long-horn beetles take 3–5 years to complete development and are about 25 mm long, and grey with minute brown spots (Ives and Wong 1988; Hiratsuka et al. 1995). Figure 2c illustrates bark damage, which usually occurs on the lower trunk, while Fig. 2d illustrates the coarse, sliver-like frass and orange to orange-brown sap ooze. Secondary infections of *Cytospora chrysosperma* are common on dead and dying trees.

Fig. 2e. *Saperda populnea moesta* LeConte (poplar gall borer) on an aspen branch. These galls are often secondarily infected by other canker pathogens, usually *Cytospora chrysosperma* in B.C., but *Entoleuca mammatum* (the cause of Hypoxylon canker) is a common secondary invader elsewhere in North America. Maturation of the grubs to mature beetles may take up to 3 years. Signs of insect activity are most evident in the fall when the larvae bore holes to the surface to expel frass (Ives and Wong 1988).

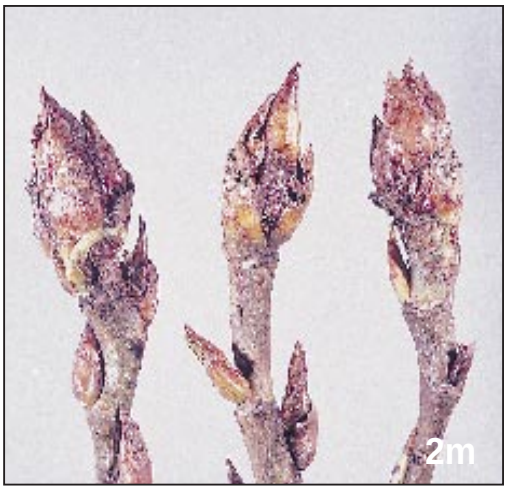
Fig. 2f. Stain, galleries, and rough bark on cottonwood caused by *Cryptorhynchus lapathi* (L.) (poplar and willow borer), a common and damaging grey-black weevil introduced from Europe in the late 1800s. *Populus trichocarpa* and many of its hybrids are favored poplar hosts in B.C., although there are a few rare records on aspen. *Salix* (the most favored host), *Alnus*, and *Betula* species may also be attacked. The weevils, which are supposed to emit a distinctive squeaking sound when handled, usually attack stems 2–8 cm in diameter. This weevil is common south of 57°N, especially on Vancouver Island, the lower mainland, and southern interior valleys (Garbutt and Harris 1994).

Fig. 2g. Aphid leaf gall caused by *Pemphigus* sp. Over 46 *Pemphigus* species worldwide are associated with different foliar and petiole galls on poplar. Some species alternate between galls on *Populus*, the primary host, and herbaceous secondary hosts such as cruciferous or umbelliferous weeds, where they colonize roots (Blackman and Eastop 1994).

Fig. 2h. Twisted petiole gall on Lombardy poplar caused by and containing *Pemphigus spyrothecae* Passerini. The life cycle of this aphid is completed on the poplar host.

Figs. 2i, j. Lace bug (*Corythucha* spp., Tingidae). Damage on hybrid poplar (Fig. 2i). The upper surfaces of affected leaves become mottled with yellowish to brownish spots. Lace bug larvae, characteristic black volcano-shaped eggs, and frass (Fig. 2j) on the foliage undersurface of a non-poplar host.

Fig. 2k. Poplar leaf pocket aphid, *Mordwilkoja vagabunda* (Walsh), on terminal shoots of trembling aspen. The secondary host of this aphid is *Lysimachia* (loosestrife). Galls, which darken to reddish brown and remain on the tree after leaf fall, protect the overwintering aphids (Hiratsuka et al. 1995).



Figs. 2l, m. "Big bud" midge (undescribed species of *Dasineura*, Cecidomyiidae) damage on T×D hybrids. Frequently encountered on *P. trichocarpa* (Gagné 1989) and many of its hybrids on Vancouver Island and the Fraser Valley.

Fig. 2n. Lygus bug (*Lygus* sp.) damage on T×D hybrid poplar, causing swollen split-stem lesions.

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Mite Stem and Branch Galls

Causal Agent

Aceria parapopuli (Keifer) (Acarina, Eriophyidae) poplar bud gall mite

Hosts/Host Specificity

Populus tremuloides, *P. trichocarpa*, rarely on T×D hybrids (one clone, 189–437, observed with leaf and bud galls), and *Populus* × *Jackii* (*P. balsamifera* × *P. nigra* hybrid) in B.C. to date. Differences in observed incidence, and size of galls in stands in northern B.C., notably near Dawson Creek (Crane and Hiratsuka 1994) suggest that aspen clones vary in susceptibility to gall mites. Aspen stands composed of several different clones may have some clones with numerous large stem galls, while other adjacent clones remain gall-free.

Time of Appearance

Mites are most active in the spring, which is when new infestations are likely to occur. Tiny reddish swellings on buds, shoots (Fig. 3b), and (rarely) leaves (Fig. 3a), are visible by mid-summer. Large perennial galls are present year-round, but are most easily observed in the winter (Fig. 3c), or in early spring prior to bud break.

Source of Inoculum

Mites spread to buds and shoots of nearby trees by crawling or by wind.

Description

Single or numerous subglobose or irregularly shaped perennial stem or branch galls, ranging in size from less than 1 cm in diameter on small branches, to more than 60 cm across on stems of mature aspen. Giant galls collected near Dawson Creek are estimated to be over 80 years old, according to Crane and Hiratsuka (1994). Galls tend to develop around the base of a developing

Distribution of *Aceria parapopuli*.

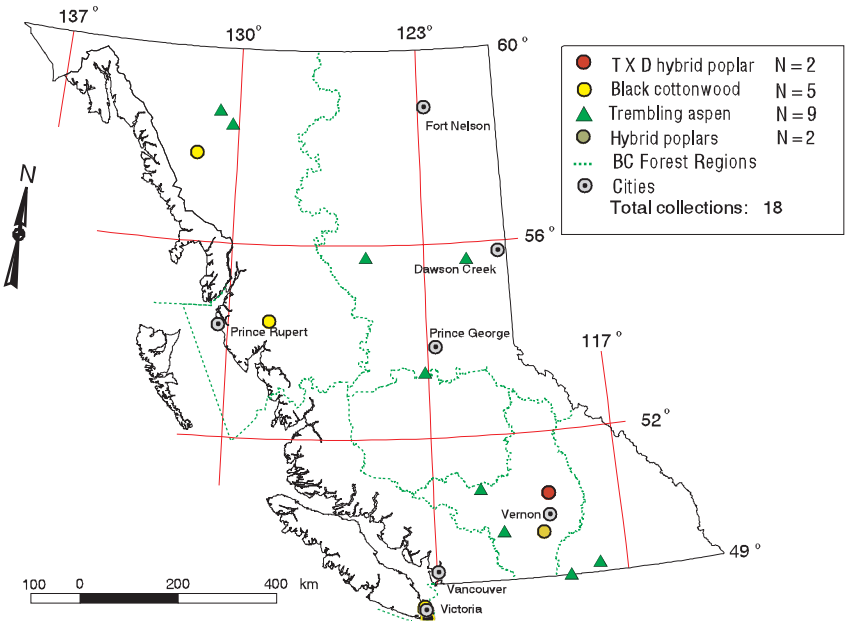

Fig 3. Poplar mite galls and black galls.

Fig. 3a. Eriophyid bud gall mite – early infection on hybrid poplar leaf.

Fig. 3b. Mite gall on aspen branch. Red lumpy areas are colonized by mites.

Fig. 3c. Large mite galls on trunk of aspen near Dawson Creek.

Fig. 3d. “Black galls” on aspen. See also Fig. 27e.

bud or shoot (Keifer et al. 1982). After several years they become woody and darkened, the surface bumpy and roughly textured like a cauliflower. Only reddish and succulent patches on the surface of large galls are currently inhabited by mites (Crane and Hiratsuka 1994). Old, blackened galls no longer contain mites (see section on look-alikes).

Key Diagnostic Features

The tiny poplar bud gall mite, *A. parapopuli*, requires a hand lens or dissecting microscope to be seen. *Aceria* spp. do not look like the typical spider mites commonly associated with webbing and foliar discoloration on many plants; the latter have eight legs, are fast-moving and spider-like. In contrast, *Aceria* bud gall mites are worm-like, never form webs, have only four short legs located near the head, and are relatively slow-moving. Poplar bud gall mites average 0.2 mm × 0.05 mm, are whitish to dark red, and usually hidden within the malformed host tissues, but may be observed slowly crawling about the reddish surfaces of the galls in the spring.

Look-alikes

The literature refers to "black galls" which also occur in western Canada (Crane and Hiratsuka 1995). Black galls are similar in appearance to mite galls (Fig. 3d), but experts have not yet confirmed the causal agent. The black galls differ from mite galls in surface and internal morphology, tending to be more globose and having a roughened, fissured surface without the finer "cauliflower" texture and reddening associated with mite activity. Researchers have observed a reduced incidence of heart rot caused by *Phellinus tremulae* in Albertan aspen with black galls (Crane et al. 1994; Hiratsuka and Loman 1984). Compounds such as benzoic and salicylic acid, present in higher concentrations in black galls, might render the galled trees more resistant to heart rot (Pausier et al. 1995). Speculations on the cause of black galls include tumor-inducing bacteria such as *Agrobacterium* (although this bacterium has never been isolated from black galls) and an abnormal growth response related to insect feeding or other physical damage.

Impact

Mite galls do not occur at levels that would cause appreciable structural or economic damage to aspen or secondary poplar hosts in B.C. Galls are considered unsightly on specimen trees in urban settings; if they occur on lateral branches, they may be removed by pruning. Areas of heavy trunk infestation in natural stands are more frequent in the northern half of the province, especially near Dawson Creek. Many of the stands infested heavily by mites also appear to have less stem decay caused by *Phellinus tremulae* (Crane and Hiratsuka 1994). Further research is required to confirm that the mite galls cause the low incidence of *Phellinus* stem decay.

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Leaf-bronzing Disease of Cottonwood and Hybrid Poplar

Causal Agent

Schizoempodium mesophyllincola Oldfield, Gispert and Hunt (Acarina, Eriophyidae) – poplar leaf-bronzing mite

Hosts and Host Specificity

T×D hybrids show a high genetic component for mite bronzing (Newcombe et al. 1995; B. Callan and P. Spencer, Canadian Forest Service, Victoria, unpublished observations). In hybrid poplar clonal trials throughout B.C., trees affected with high levels of leaf bronzing frequently intermesh canopies with unaffected T×D clones. Current studies (A. Hunt, University of Victoria, pers. comm.) aim to determine if leaf morphology influences susceptibility to colonization and bronzing by mites. *Populus trichocarpa* leaves have a characteristically spongy, open mesophyll layer below two layers of a palisade mesophyll. In contrast, *P. deltoides* leaves have a compact "double palisade" of mesophyll cells, and have no layer of spongy mesophyll which contains large intercellular air spaces (beautifully illustrated in Van Volkenburgh and Taylor 1996). Leaf structure of T×D hybrids is variable – some hybrid clones have intermediate morphological forms, while others look more like one parent than the other. T×D hybrids with leaf morphology closer to that of *P. deltoides* appear to be more resistant to leaf bronzing, and have a darker green leaf undersurface. T×D hybrids with leaf morphology closer to that of *P. trichocarpa* usually have whitish leaf undersurfaces; the paler color and higher reflectivity are due to the air spaces in the mesophyll (Van Volkenburgh and Taylor 1996). This looser mesophyll structure appears to permit greater ease of mite movement within the leaf.

Data collected from clonal adaptiveness trials surveyed on northern Vancouver Island from 1995 to 1997 showed that approximately two-thirds of the trees had some level of mite infestation (B. Callan and P. Spencer, unpublished observations). *Populus deltoides* is resistant, and *P. deltoides* parents confer resistance to some of the T×D hybrids (pers. comm. George Newcombe, Washington State University, Puyallup). Susceptible T×D clones are often more severely bronzed than adjacent pure *P. trichocarpa* of the same age. Some T×N and T×M clones were also highly susceptible, while native *P. trichocarpa* showed only moderate bronzing levels. M × M clones appear to be resistant (B. Callan and P. Spencer, unpublished observations). *Populus tremuloides*, *P. nigra*, and *P. alba* appear to be resistant. Susceptibility of other *Populus* species is unknown.

A correlation has been observed between low levels of foliar rust infections by *Melampsora medusae f. sp. deltoidae* on leaves bronzed by mites (Newcombe et al. 1995). This correlation, regardless of whether the mite causes foliar damage itself, could be a useful additional tool for field assessment of rust susceptibility in years of low rust incidence.

Time of Appearance

Poplar leaf bronzing disease was first discovered in 1989 in hybrid poplar clonal trials on northern Vancouver Island, near Sayward, B.C. (Oldfield et al. 1998). Bronzing symptoms first become apparent on foliage in late June.

Source of Inoculum

Deutogynes (overwintering females) cluster on the most recent year's twigs in sequestered sites (hibernaria) such as lenticels, bud scales, or old insect wounds on young branches. They emerge in May and June to colonize new foliage, moving through leaf stomata into the spongy mesophyll, where they feed and lay eggs. The immature stages, including eggs, larvae and nymphs, are only found in the mesophyll of susceptible leaves. Adult mites may be seen moving

about on the surface of heavily infested leaves. The population of mites within bronzed leaves drops drastically before leaf fall from September to early October, when mites begin to leave the foliage in search of overwintering sites (Hunt 1998).

Description

The common name for the disease is derived from the symptoms on the abaxial (lower) sides of the leaves, where at first small angular areas of mesophyll (5–25 mm^2) become discolored brown, and subsequently coalesce with adjacent bronzed areas. Bronzing first becomes visible at the base of the leaf blade, and expands along the edge, mid-rib, and major veins. Eventually the whole leaf undersurface becomes bronzed or even blackened in extremely susceptible hosts. The upper (adaxial) surface of the leaves remain more or less green.

The undersurface of bronzed leaves collected from mid-June to mid-September should be examined under the highest magnification of a dissecting microscope, preferably by transmitted light (a bright light shone through the leaf from underneath). Examination of bronzed leaves reveals minute (about 0.2 mm long × 0.03 mm wide) worm-like eriophyoid mites crawling over the surface and inside bronzed regions of the leaf mesophyll. In September, numerous mites should also be visible moving around on the leaf undersurface. Mites may not be visible early in the growing season, when the population is low. Deutogynes should be visible in masses in hibernaria on dormant susceptible trees which were infested the previous year. Mites are not present on symptomless foliage. Late in the growing season, the number of mites in bronzed leaves is reduced due to migration from foliage to overwintering sites elsewhere on the tree (Hunt 1998).

Key Diagnostic Features

- bronzed undersurface of otherwise green leaf
- presence of large numbers of eriophyoid mites within mesophyll

Impact

The causes of foliar bronzing, whether it is toxins produced by the mite, physical damage due to feeding, or some other factor, is currently unknown. The impact on growth and yield is expected to be low.

Fig. 4. Poplar leaf-bronzing mite.

Fig. 4a. Bronzed leaf compartments indicate areas infested by mites.

Fig. 4b. Bronzing patterns on leaves, illustrating development from the edges and main veins.

Fig. 4c. Hybrid (T×D) poplar leaves, green on the upper surfaces but completely bronzed underneath.

Fig. 4d. Blackening of infested hybrid poplar (T×D) leaves.

Fig. 4e. Hundreds of cream-colored worm-like overwintering leaf-bronzing mites clustered at a T×D leaf scar (epidermis removed × 25).

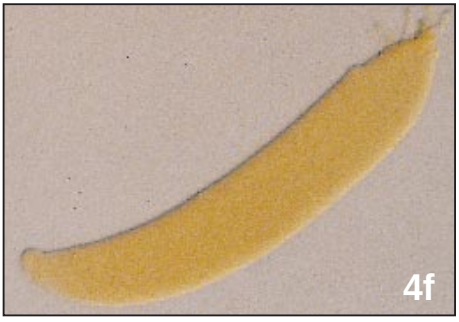

Fig. 4f. Deutogyne (overwintering female) of *Schizoempodium mesophyllincola*. Photomicrograph (× 360).

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Feeding by vertebrates (Fig. 5) occasionally damages all species of poplar in B.C., and may reach severe levels in cottonwood and hybrid poplar plantations. Just as with fungal pathogens and insects, preference for certain clones over others is often noticeable, both in natural stands and in plantations (Dickmann 1978).

Harvesting of hardwood stands increases the likelihood of feeding damage from ungulates, rodents, and birds, who often prefer to graze on the shoots and bark of saplings at clearing edges. In various regions throughout the United States, research has shown that biomass in regeneration which serves as food and shelter for rodents increases in availability following clearcutting of deciduous stands (Kirkland 1977; Conroy et al. 1979). In B.C. this phenomenon is certainly observed in areas of natural regeneration, and also in newly established hybrid poplar plantations. Prior to canopy closure, young saplings are frequently surrounded by dense growth of grasses and other herbs, which provide protection from predators. Vole damage may be extensive in such areas, especially if weeds are not controlled, or if protective collars are not wrapped and maintained around the young stems until canopy closure shades out the weeds.

Most poplars are not killed by vertebrate feeding damage but wounds act as infection courts for decay, stain, root disease, and canker fungi, as well as insects. Repeated clipping of new growth also results in poor form. A good general review of the relationship of wildlife to aspen has been written by DeByle (1985).

Fig. 5. Vertebrate damage.

Fig. 5a. Beaver (*Castor canadensis* Khul.) damage of trembling aspen occurs in riparian areas. Felled aspen are used extensively in lodge and dam construction (Enns et al. 1993).

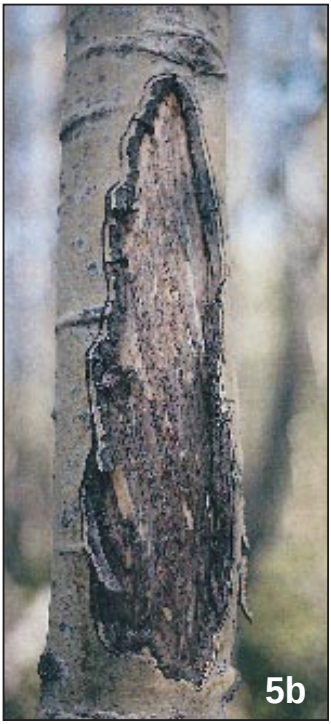

Fig. 5b. Bear (*Euarctos* [grizzly] and *Ursus* [black] spp.) damage on trunk of trembling aspen, with canine teeth and claws creating parallel grooves and long bark shreds. Feeding occurs on sapwood during spring and summer (Harestad et al. 1986).

Fig. 5c. Snowshoe hare (*Lepus americanus* Erxelben) damage on young poplars. This animal is considered second only to fire in retarding new growth of aspen succession in central Canada (Bird 1930). Areas with chewed bark are characterized by ragged parallel tooth marks about 2 mm across (Harestad et al. 1986). Damaged areas may reach almost 1 m above ground, depending on the depth of snow cover surrounding the tree trunk. Buds and shoots may often be clipped off at an acute angle, although the resin of balsam poplars is unpalatable (Peterson et al. 1996). Hare populations cycle to a peak number every 9–10 years. Snowshoe hares are not present on Vancouver Island. Rabbits (*Lepus* spp.) cause similar damage.

Fig. 5d. Moose (*Alces alces* Gray) feeding scar (parallel grooves of healed bark) on trembling aspen. Feeding damage, caused by upward scraping of incisors, most commonly occurs in early winter.

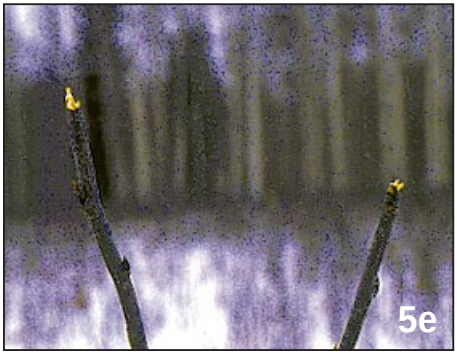

Fig. 5e. Deer (*Odocoileus* spp.) browse on aspen saplings causes ragged, squared edges on the chewed shoots. Elk (*Cervus* spp.) and moose browse causes similar damage. Patches of bark on small trees may also be stripped in the fall when they are used to polish antlers.

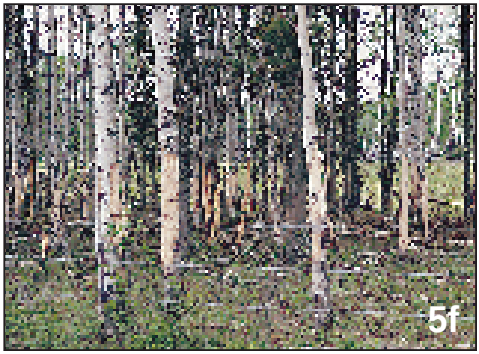

Fig. 5f. Cattle-damaged trembling aspen.

Fig. 5g. Vole (*Microtus* spp.) damage at the base of a T×D hybrid poplar, with irregular patches of bark removed from trunk and roots, and the exposed wood "fuzzy" from many tiny (1.5 mm wide) tooth marks. Vole damage is more likely to occur in areas with heavy ground cover.

Fig. 5h. Sapsuckers (*Sphyrapicus varius varius* L. in the interior and northern B.C.; *S. varius ruber* (Gmelin) on the coast, and exclusively on Vancouver Island) cause wounds on trembling aspen which form characteristic parallel lines of bark holes. The bird returns to the wounded tree over extended periods to feed on the sap oozing from the holes, and on insects attracted to the sap (Ziller and Stirling 1961). Trees at the edge of logged sites or plantations are preferred, and the wounds may become an entrance site for insects, and fungi that cause decay, cankers, and stain. Hybrid poplars, *Betula*, and *Pinus* species are also attacked by sapsuckers (Hiratsuka et al. 1995).

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A canker is a sunken necrotic lesion of the tree's stem or branch caused by localized death of bark. Cankers on the main stems may eventually kill the tree by girdling. They also greatly increase the chance of stem breakage due to irregular radial growth, or by opening heartwood to decay organisms (Callan 1997). The value of lumber from cankered deciduous and coniferous trees may be reduced by discoloration and abnormally high proportions of nonwoody cells, which interfere with appearance, wood strength, and preservative penetration. Pulp value is also lowered by increased difficulty in the bleaching of discolored wood in cankers, debarking problems, and high levels of wood fibres and stained chips in cankered wood (Baranyay et al. 1973; Nevill et al. 1989).

The extent and severity of a canker depends on variables such as environmental conditions, species of pathogen, host response, as well as size, age, and time of year of host tissue infection. Many types of cankers are caused by facultatively parasitic fungi which require a wound or other host stress to become established. Other cankers are caused by environmental factors such as freezing or drought.

Most cankers can be defined in the following general categories:

1. Perennial or target cankers: generally circular to lens-shaped cankers that persist for years, and slowly expand at about the same rate as the radial growth of the affected tree. Perennial cankers gradually take on a sunken appearance, as tissues under the dead cambium do not grow along with the surrounding wood. The cankers are often surrounded by concentric ridges of host callus tissue formed during each growing season in response to the infection, resulting in a target-like pattern. In rapidly growing trees, the callus may eventually grow over and cover infected areas. *Ceratocystis* spp. are associated with perennial target cankers of aspen, for example.
2. Diffuse cankers: cankers that expand more rapidly than the radial growth of the tree and so include little or no callus. Rapid growth of diffuse cankers often girdles the tree in a few years. Diffuse cankers are more commonly encountered in young tissues invaded by a fungal pathogen, such as young poplars with *Cytospora*, *Neofabraea*, and *Cryptosporiopsis* cankers.
3. Annual cankers and twig blights: cankers that also contain little or no callus, but usually develop rapidly and for one season only. They frequently occur on young stems or twigs that have been predisposed to opportunistic fungal infections by frost, drought, other stresses, or wounding. The dead bark may be overgrown by callus in the following growing season, resulting in complete healing if the external stresses are no longer present. Under the conditions of extreme moisture found in coastal hybrid poplar plantations on Vancouver Island, *Venturia populina* may be associated with annual cankers. *Cytospora chrysosperma* also causes twig blights, especially on shade-stressed branches.

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Key to Common Cankers and Bark Diseases

1. Bark rough, swollen, corky, but alive; not directly associated with foliar distress 2
1. Bark sunken, dead; trees often showing signs of foliar distress on branches directly above or adjacent to cankered side of trunk or branch 3

2. Bark with swollen, corky, longitudinally ridged regions extending from a few centimeters to several meters along the trunk of aspen . . . . . *Rhytidiella baranyayi* (*R. moriformis* on rough-bark of balsam poplar)
2. Bark not longitudinally ridged, but rounded, tumored, and gall-like; bearing black apothecia (0.5–1.0 mm in diameter) or acervuli (0.5 mm in diameter) . . . . . *Leciographa gallicola*

3. Dead bark surrounding one or several holes or wounds, which are either surrounded by sawdust-like frass, or oozing an orange or brown, foul-smelling flux . . . . . .wood-boring insect activity, followed by bacterial wetwood
3. Dead bark not associated with insect wounds, frass, or slime flux 4

4. Fungal fruiting bodies absent from cankers or the surrounding bark 5
4. Fungal fruiting bodies consistently developing on cankers or the surrounding bark 6

5. Damage occurring during dormant season (examine growth rings), consistently on the same, exposed side of all trees affected . . . . . thaw/freeze winter damage
5. Damage occurring during summer, on sunny side of trees adjacent to roads or newly exposed trees at edges of cutblocks or thinning operations . . . . . sun-scald

6. Cankers discrete, sunken, concentrically zonate (perennial); always on aspen . . . . . *Ceratocystis* canker
6. Cankers more diffuse, not concentrically zonate, and on various *Populus* spp. 7

7. Blackened discoloration and bark pimpling of cuttings (cottonwood and hybrids) predisposed by frost, drought, or shade (blackstem disease complex) 8
7. Not as in preceding description 9

8. Blackstem associated with orange spore tendrils or ooze . . . . . *Cytospora (Valsa sordida)*
8. Blackstem associated with white spore tendrils or ooze . . . . . *Phomopsis (Diaporthe eres)*

9. Cankers causing abundant rough cracks throughout affected bark, with fungus tissue forcing bark to raise away from wood; only on aspen 10
9. Cankers smoother, diffuse, usually only cracked at the margin; forming dead discolored longitudinal regions; fruiting bodies often pimpling surface of dead bark, on various poplars 11

10. Cankers large, up to several meters long, diffuse, faintly zonate, sooty, black, on standing, overmature aspen; producing abundant grey, granular apothecia, 1–3 mm in diameter, on fallen trees . . . . . .Sooty-bark canker, caused by Encoelia pruinosa
10. Cankers smaller (usually less than 0.5 m long), crusty, grey, and never zonate; at first with dusty columns of fungal tissue forcing bark away from cambium then exposed areas bearing a layer of grey perithecia . . . . . .Hypoxylon canker *(Entoleuca mammata)*

11. Affected bark discolored orange, becoming coarsely pimpled with fruiting bodies which contain or exude slimy masses of orange spores; fruiting bodies appearing highly convoluted when sectioned (or in mature specimens, clusters of round perithecia opening at a central ostiole which may be ringed with white) . . . . . .Cytospora canker *(Valsa sordida)*

11. Not as in preceding description, or spore masses or tendrils not orange 12

    12. Spore masses white to pale yellow, erumpent in acervuli; brown cushion-like apothecia developing in same areas as the acervuli, primarily on cottonwood, balsam, and hybrid poplars . . . . . .Neofabraea canker

    12. Spores pale yellow-brown, not exuding in tendrils 13

13. Extensive (up to several meters long), smooth to lightly roughened areas of tan-colored dead bark thickly dotted with black ostioles, which when cut reveal circular black fruiting bodies about 0.4 mm in diameter underneath . . . . . *Cryptosphaeria populina*

13. Small sunken cankers on young stems, usually connected to a blackened, blighted shoot or petiole; fruiting bodies not evident, but dead tissue may be covered in places with olive green spore masses . . . . . *Venturia populina*

Blackstem Disease of Hybrid Poplars and Cottonwood

Causal Agents

Valsa sordida Nitschke (Ascomycetes, Diaporthales) (anamorph = *Cytospora chrysosperma* (Pers.:Fr.) Fr.). See section on Cytospora canker for full description.

Diaporthe eres Nitschke – (Ascomycetes, Diaporthales) (anamorph = *Phomopsis oblonga* (Desmaz.) Traverso)

Hosts/Host Specificity

In B.C., blackstem is associated with stool beds and newly planted cuttings of *P. trichocarpa* and its hybrids. Throughout North America both species of fungi are found on native and introduced poplars and their hybrids.

Time of Appearance

Blackstem appears after stress caused by drought, insect attack, sunscald, repeated defoliation due to disease, frost, or nutrient deficiency.

Source of Inoculum

Wind-spread ascospores and water-splashed conidia provide inoculum, which enters bark through wounds, bud scales, or leaf scars (Ostry et al. 1988).

Description

Cytospora canker (Figs. 6a, b) is described elsewhere. Blackstem caused by *Phomopsis* looks superficially similar, turning the bark first orange, then black, and producing pimple-like eruptions on the bark surface (Fig. 6c). However, spore tendrils and masses are white to cream-colored rather than orange. Pycnidia of *Phomopsis oblonga* are erumpent from the epidermis, 1–2 mm in diameter and produce two distinct types of conidia: alpha conidia are hyaline, single-celled, and ellipsoid, 6–9 × 2–3 µm; beta conidia are hyaline, single-celled and thread-like, 25–33 × 1 µm (Fig. 6d). The teleomorph, *D. eres*, produces perithecia in the same areas as the pycnidia. Perithecia are up to 1 mm in diameter, and often loosely grouped with necks converging to a single point on the blackened bark surface. Ascospores are hyaline, two-celled, constricted at the central septum, 10–14 × 2.5–4 µm (Funk 1981).

Key Diagnostic Features

- bark of cuttings or stems in stool beds turning first orange then black
- failure of cuttings to sprout
- pimpled bark surface, with orange or white spore tendrils extruding from pimples in humid weather

Look-alikes

Blackening of drought or diseased-stressed T×D hybrid poplar shoots may also be caused by the opportunistic blight fungus *Colletotrichum gloeosporioides* (Penz.) Penz. and Saccardo. This fungus first produces slimy pinkish masses of conidia in acervuli on blackened tissues (Fig. 6c). The acervuli are usually surrounded by black setae, and the conidia are hyaline, and irregularly cylindrical to ellipsoidal, measuring 12–30 × 4–6 µm. Later, small (0.1–0.3 mm in diameter), browinish perithecia of the sexual state, *Glomerella cingulata* (Stoneman) Spauld. and H. Schrenk, develop on dead tissues. Asci are cylindrical to ellipsoid, 35–80 × 8–14 µm, containing four to eight hyaline, single-celled, ellipsoid ascospores measuring 12–28 × 4–7 µm (Funk 1981). Newcombe et al. (1991) also reported *G. cingulata* associated with Venturia shoot blight of hybrid poplars.

Impact

Unrooted cuttings can be damaged by blackstem during storage or shipping, especially if they were taken from trees or stool beds prone to blackstem.

Fig. 6. Blackstem disease.

Fig. 6a. Blackstem disease caused by *Cytospora* on T×D hybrid poplars following drought stress.

Fig. 6b. Orange *Cytospora* conidial tendrils emerging from shaded T×D hybrid poplar stems with blackstem disease.

Fig. 6c. Blackstem (caused by *Cytospora* and *Phomopsis*) on newly planted, drought-stressed T×D hybrid poplar cuttings.

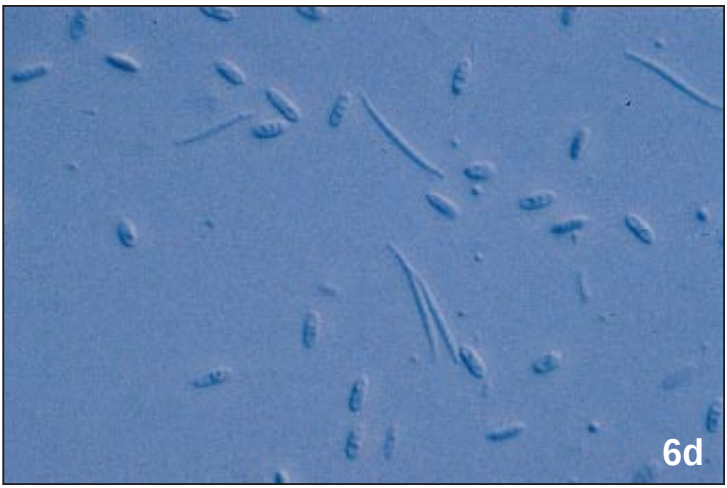

Fig. 6d. Alpha and beta conidia of *Phomopsis oblonga*. Photomicrograph (× 1250).

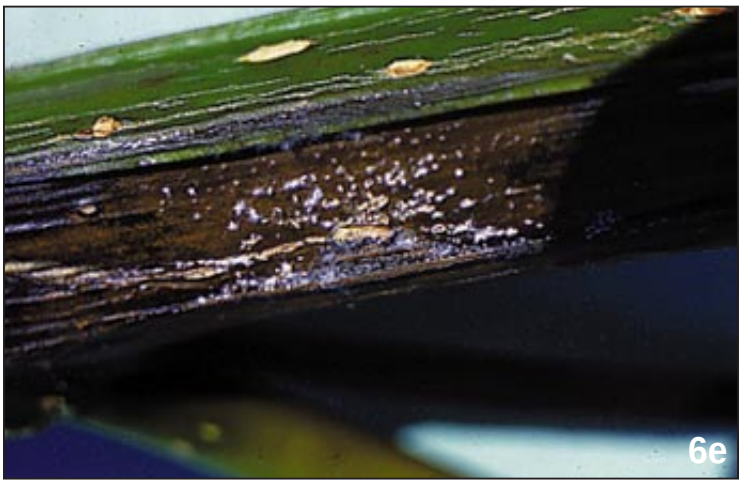

Fig. 6e. Pink spore masses on a T×D hybrid poplar infected with *Colletotrichum*.

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Cytospora Canker

Causal Agent

Valsa sordida Nitschke (Ascomycetes, Diaporthales) (anamorph=*Cytospora chrysosperma* (Pers.:Fr.) Fr.)

Hosts/Host Specificity

All native, introduced and hybrid poplars throughout their range are hosts. *Salix, Acer, Alnus, Prunus*, and other deciduous trees are also known as occasional hosts in B.C. and elsewhere in North America.

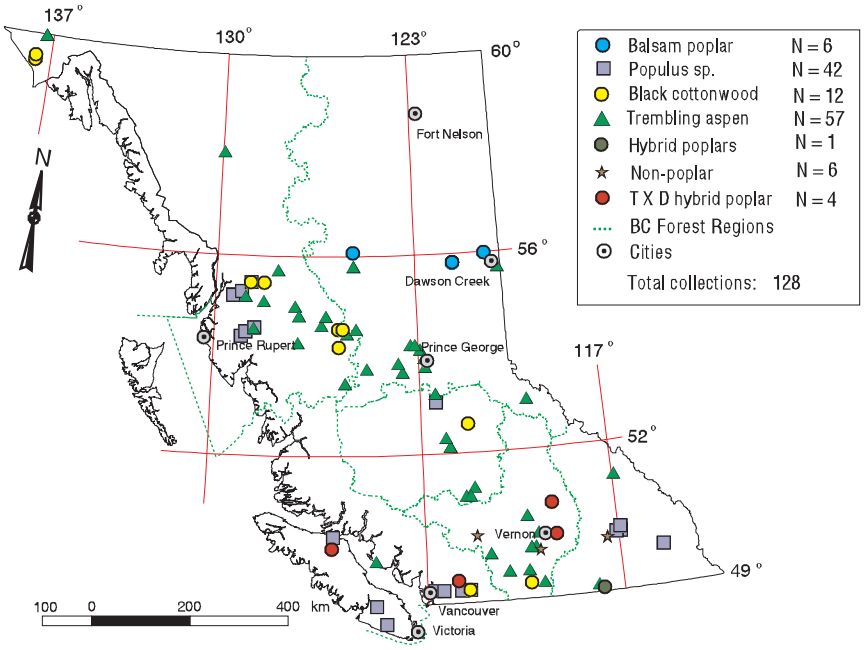
Time of Appearance

Initial bark discoloration (Fig. 7a) appears several weeks after the host cambium has been damaged by agents such as drought, frost damage, sunscald (Fig. 7b), wounds, or primary pathogens (see section on Cryptosphaeria canker).

Source of Inoculum

Cytospora cankers do not occur on healthy, vigorous, undamaged trees. Predisposition via wounding, fire, drought, or other damaging agents is required for infections to be successful. The cankers have two sources of inoculum: conidia, which extrude in sticky tendrils, are spread by rain and dripping water; or ascospores, which are forcibly ejected from the perithecia and become airborne. Young plantations and stool beds may become infected by spores produced on branch or stem cankers on adjacent, overhanging mature trees, or by diseased propagation material (see section on blackstem disease).

Distribution of *Valsa sordida*.



Description

Infected bark discolors to orange or orange-brown, and eventually darkens to black. Pycnidia are immersed in the bark, with ostioles pushing through the surface, causing it to be roughly pimpled (Fig. 7c). Pycnidia (Fig. 7e) range from 0.5 to 1 mm in diameter with bases folded into a convoluted set of locules (Fig. 7f) lined inside with filamentous, often branched conidiogenous cells 10–40 µm long × 1 µm in diameter (Fig. 7g). Conidiogenous cells bear phialides which produce hyaline, sausage-shaped, unicellular conidia averaging 3–5 × 1–1.5 µm. Masses of conidia are produced during periods of damp weather (or after excised bark is incubated in a moist chamber), and may extrude for several centimeters in orange to reddish orange tendrils from the pycnidial ostioles (Fig. 7d).

Perithecial stromata form after pycnidia, often replacing them in the same cankered locations on the bark. Stromata are conical, 0.5–2 mm in diameter, with ostioles breaking through the bark and surrounded by a pale to dark grey disc. Stromata contain a cluster of 6–12 globose, 0.3–0.5 mm in diameter, dark greyish brown perithecia, whose long narrow necks collectively open into the ostiole. Asci are club-shaped, 30–45 × 5–7 µm, eight-spored (Fig. 7h), and wider at the apical end which is flattened and bears a hyaline ring (does not stain blue in Melzer's reagent). Asci detach from perithecial wall and float free within the perithecium when mature. Ascospores are hyaline, allantoid, single-celled, 7–12 × 1.5–2.5 µm.

Fig. 7. Cytospora canker of poplars.

Fig. 7a. Typical orange discoloration of Cytospora canker, following physical damage of aspen trunks.

Fig. 7b. *Cytospora* infection following sunscald damage of aspen.

Fig. 7c. Bark discoloration, and pycnidia of *C. chrysosperma* on a T×D hybrid poplar trunk.

Fig. 7d. Orange conidial tendrils of *C. chrysosperma* are extruded during periods of high humidity (× 4).

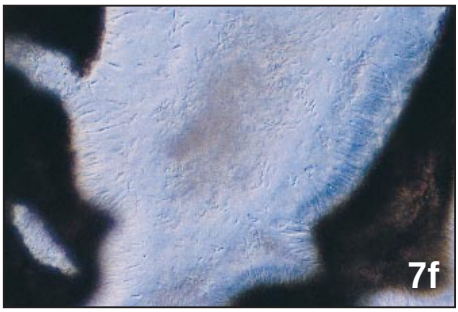


Fig. 7f. Section of pycnidium, illustrating dark convoluted pycnidial wall, lined by a fringe of conidiogenous cells, and filled with a pale orange mass of conidia. Photomicrograph (× 100).

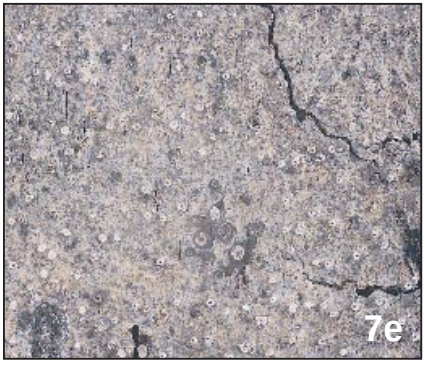


Fig. 7e. White-rimmed pycnidial ostioles emerging from cankered aspen bark (× 5).

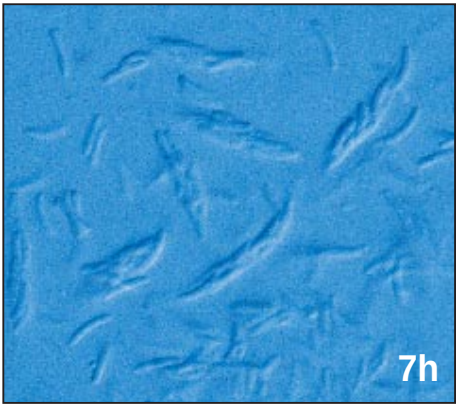


Fig. 7h. Asci and ascospores of *Valsa sordida*. Photomicrograph (× 600).

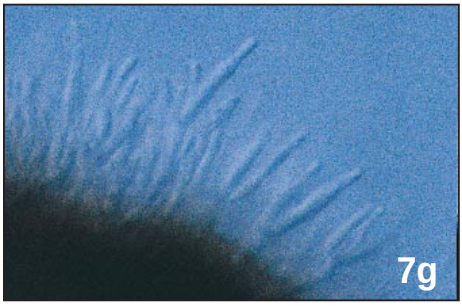


Fig. 7g. Detail of conidiogenous cells and developing conidia. Photomicrograph (× 1000).

Key Diagnostic Features

- orange to reddish orange spore tendrils, which may reach lengths of 10 cm
- cankered areas and infected bark turning bright orange to orange-brown, and finally black
- fruiting bodies (pycnidia, perithecia) causing bark to become roughly pimpled
- pycnidia with convoluted bases and orange to red-orange contents

Impact

Cytospora chrysosperma is the most common opportunistic canker-inducing pathogen in B.C. As early as 1931, it was documented causing cankers on 11 different fire-damaged hosts (Salicaceae, including *Populus* spp., Betulaceae, Rosaceae, Cornaceae, Aceraceae, and others) following a light ground fire approximately 130 km north of Vancouver (Dearness and Hansbrough 1933).

Once the fungus colonizes and girdles a damaged main shoot or branch, the resulting desiccation of the affected tissues further stimulates fungal growth and sporulation. Dieback of poplars will occur in early spring if plants set terminal buds too late the previous fall. The combined effect of solar radiation followed by secondary *Cytospora* infection (Bloomberg 1962) causes the injury (Bloomberg 1962).

Cytospora species also play an important role in the succession of decay in dead aspen, based on the relative frequency of isolation from branches of different ages (Tao et al. 1984).

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Sooty-bark Canker

Causal Agent

Encoelia pruinosa (Ellis & Everh.) Torkelson & Eckblad (Ascomycetes, Helotiales)

Hosts/Host Specificity

Known only from trembling aspen in B.C. Widespread on this host and other related aspen species throughout North America and Europe. Black cottonwood and balsam poplar have been reported to be occasionally infected (Sinclair et al. 1987).

Time of Appearance

Cankers appear after wounding of the bark, at any height of the trunk. Higher incidence of cankers could be expected after stand disturbance events such as thinning or selective logging, or in high-use areas such as campsites. Large (1 m long) cankers may develop within 1 year of infection.

Source of Inoculum

The inoculum source is presumed to be ascospores, which are forcibly ejected and airborne when bark moisture levels are sufficiently high to ensure the apothecia are fully expanded.

Description

Cankers expand rapidly after infection and measure up to 1 m × 35 cm in the first year (Hinds and Ryan 1985). Young infections appear as sunken elliptical regions with underlying blackened cambium. This inner blackened layer remains attached to the tree after the outer bark has sloughed off, thus giving the sooty appearance to the canker. Large, older cankers are often indistinctly zonate, and each zone marks a year of expansion (Fig. 8a). The edges of the cankers in susceptible trees are not noticeably raised, as the tree cannot form defensive ridges of callus tissue each year.

Distribution of *Encoelia pruinosa*.

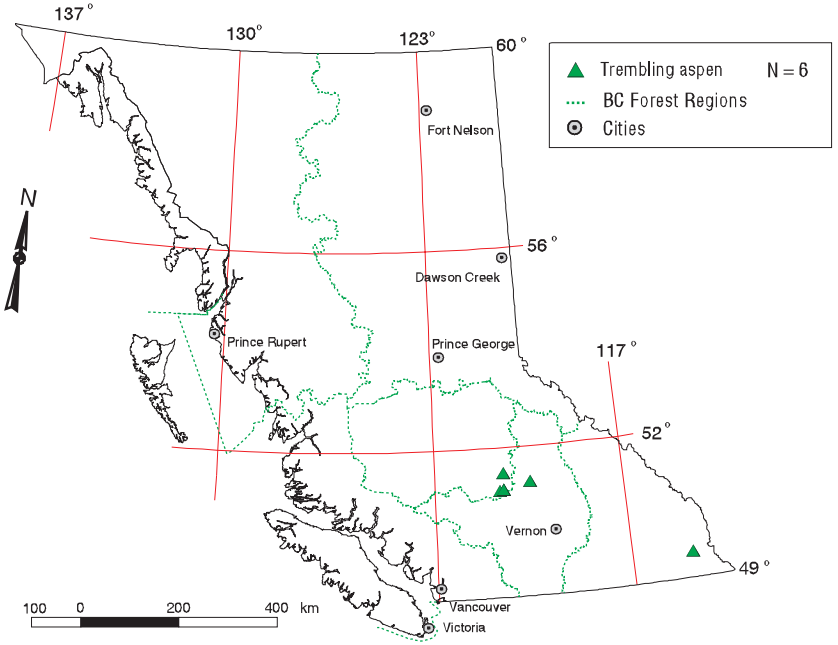

Fig. 8. Sooty-bark canker on aspen.

Fig. 8a. Large trunk canker on dying mature aspen.

Fig. 8b. Dried, curled grey apothecia clustered on exposed canker surface of dead aspen.

Fig. 8c. Apothecia (× 2).

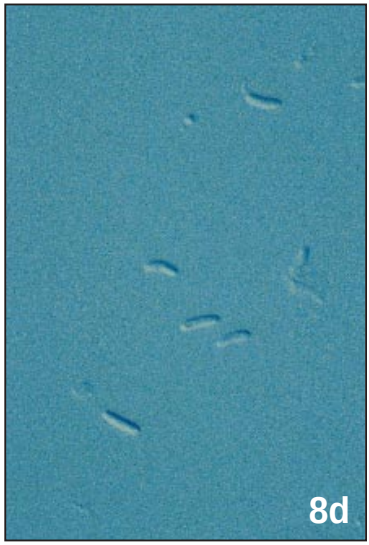

Fig. 8d. Ascospores. Photomicrograph (× 500).

The short-stalked, saucer-shaped apothecia, which range from 1–3 mm in diameter, erupt by the thousands in blackened areas of standing and fallen dead trees where bark has sloughed off (Fig. 8b). When the weather is dry, the outer apothecial margins curl inward, and the apothecia roll into greyish brown, rough-surfaced, longitudinally split tubes (Fig. 8c). When the weather is damp, the tubes unroll to expose the brown hymenial surface, which bears the asci. The club-shaped asci average 40–55 × 4–5 µm, and contain eight spores and a small apical plug which stains blue in Melzer's reagent. Ascospores are sausage-shaped, 8–11 × 2–3 µm, and hyaline, and rarely bud within the ascus (Fig. 8d) (Davidson and Cash 1956).

Key Diagnostic Features

- black, sooty residue, often in a spotted pattern, on old cankered bark
- long, often spirally twisting, roughly zonate, black cankers up to 3–4 m long on mature or overmature aspen
- thousands of small, grey-brown, granular apothecia on blackened dead snags and fallen trees in areas where cankers are common

Impact

Sooty-bark canker is the most damaging primary canker pathogen of aspen in northern and central B.C., and in the Rocky Mountain region of the United States (Juzwik et al. 1978). It has not been collected on Vancouver Island or coastal B.C., and does not appear to be associated with perennial cankers in eastern North America. In areas where they are common, cankers can girdle mature to overmature aspen just a few years after becoming established, expanding horizontally in some trees at a mean rate of 16.3 ± 0.7 cm/year, growing to lengths of 1 m in the first year of infection (Hinds and Ryan 1985). These expansion rates were based on aspen measured in Colorado; girdling rates may be faster or slower in B.C., but this figure could enable forest managers to estimate the remaining number of years until tree death. Trees die within a year after they are girdled, but leaves may still flush the spring after the tree is girdled. In Colorado, more than half the tree mortality in commercially managed aspen stands is attributed to sooty-bark cankers (Hinds and Ryan 1985). This disease is uncommon in aspen stands younger than 60 years old (Davidson and Cash 1956). Scattered individual stands of mature aspen may be heavily affected, with a large percentage of stems cankered. This pattern might indicate clonal variation in disease susceptibility (Juzwik et al. 1986).

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Ceratocystis Canker

Causal Agent

Ceratocystis fimbriata Ellis & Halst. (Ascomycetes, Ophiostomatales)

Hosts/Host Specificity

Ceratocystis fimbriata has been shown to be the causal agent of cankers of *P. tremuloides* in B.C. and elsewhere in North America (Kile 1993; Wood and French 1963). It has also been isolated from shoots of hybrid poplars in *Populus* sections *Tacamahaca* and *Aigeiros* in Poland. *Ceratocystis fimbriata* is also pathogenic to a variety of other plant hosts, including *Ipomoea* (sweet potato), *Prunus*, and *Quercus.*

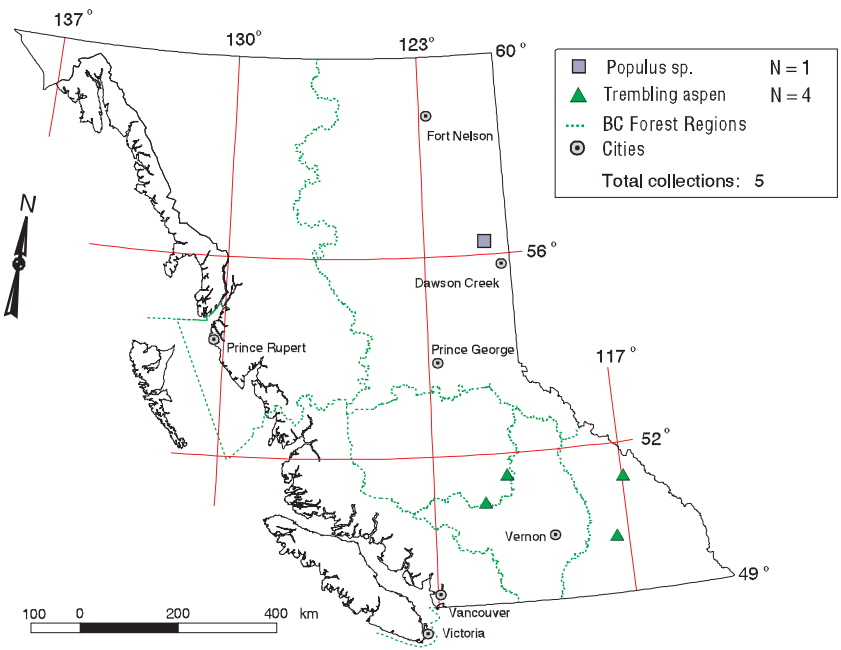
Time of Appearance

Infections develop on recently wounded host tissue, after it is visited by insect vectors a few days to a few weeks after the damage occurs (Hinds 1972b). Many cankers are centered around old branch stubs. Infections may also progress from wounded leaves and petioles to young twigs, as well as developing directly on mature trunks at wounds caused by insects, fire scars, or other types of physical damage (Hinds 1972a; Zalasky 1965b). Perennial target cankers may therefore be detected at any time of the year, and may be as old as 40–60 years (Hinds and Ryan 1985).

Source of Inoculum

Many different insects are known to be vectors of *C. fimbriata,* specifically beetles and some fly species. Studies in Colorado (Hinds 1972b) have shown that these insects are attracted to freshly wounded trees.

Distribution of *Ceratocystis fimbriata.*



Description

Cankers (Fig. 9a) are oval to lens-shaped, up to 0.5 m long, concentrically ridged, with sunken centres. Cankered bark is blackened, and may take on a varnished appearance due to orange dried sap exudations.

The black, long-necked perithecia are embedded or protruding from the cankered bark and exposed sapwood (Fig. 9c), 0.1–0.2 mm in diameter, with necks as long as 0.8 mm, often appearing as black bristles. Asci are short-lived and globular. Ascospores are hyaline, sticking together in waxy clumps at tips of perithecial necks, and when mounted in water, 3–5 × 6–8 µm, shaped rather like a bowler hat (round, but one side flattened, with the edge extended to form a "brim") (Fig. 9d). The fungus produces a distinctive *Chalara* anamorph, which forms cylindrical single-celled conidia measuring 10–27 × 3–5 µm, borne within cylindrical conidiogenous cells (phialides) (Zalasky 1965a).

Look-alikes

Other species of *Ceratocystis* may be also present on the cankered tissues, but *Ceratocystis fimbriata* is the only species considered a causal agent of cankers in B.C. It is distinctive in its *Chalara* anamorph, and hat-shaped ascospores. *Ceratocystis crassivaginata* Griffin produces fusiform ascospores in small (0.04–0.09 mm in diameter) perithecia and *Ceratocystis tremuloaurea* Davidson & Hinds is characterized by 0.09–0.15 mm in diameter perithecia and crescent-shaped ascospores (Funk 1981).

Key Diagnostic Features

- perennial, slowly expanding, elliptical zonate canker; only on aspen
- presence of *Ceratocystis* perithecia greater than 0.2 mm in diameter on the ridges of the cankered wood

Fig. 9. Ceratocystis canker of trembling aspen.

Fig. 9a. Perennial target canker.

Fig. 9b. Stem breakage at canker, often associated with secondary decay fungi.

Fig. 9c. Perithecial necks protruding from surface of cankered wood (× 13).

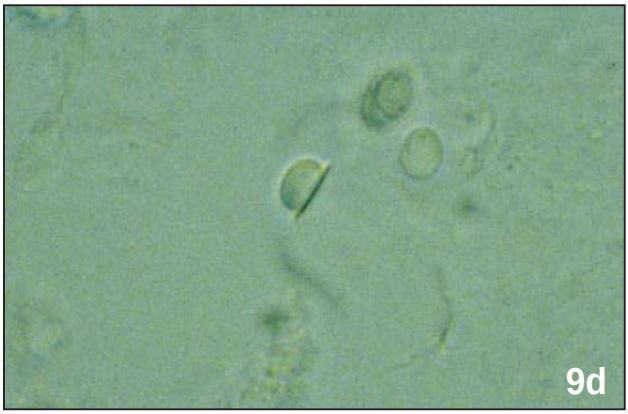


Fig. 9d. Ascospores, shaped like bowler hats. Photomicrograph (× 1500).

Impact

Ceratocystis cankers are rarely reported from B.C., but they result in severe girdling damage to the affected trees. Cankered stems are also prone to wind breakage (Fig. 9b).

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Cryptosphaeria Canker

Causal Agent

Cryptosphaeria lignyota (Fr.:Fr.) Auersw. (Ascomycetes, Diatrypales) = *Cryptosphaeria populina* (Pers.) Sacc. (anamorph = *Cytosporina* (= *Libertella*))

Hosts/Host Specificity

In B.C., Cryptosphaeria canker has been reported from *P. tremuloides* and *P. trichocarpa*. Elsewhere in North America, hosts include *P. grandidenta*., various aspen hybrids, *P. deltoides*, Lombardy poplar (*P. nigra* L. cv. "*italica*"), and purple willow (*Salix purpurea* L.). It has also been reported from Europe on *P. nigra* and other poplars (Ellis and Ellis 1985). Cryptosphaeria canker of aspen has been studied most extensively in the northern United States, where in some areas, such as Colorado, it is the second most common canker disease after Hypoxylon canker (Juzwik et al. 1978).

Time of Appearance

The cankers are usually associated with trunk or branch wounds. Perithecia develop in bark which has been dead for at least 1 year, and sporulate from June to September (Ellis and Ellis 1985). Artificial inoculation studies (Hinds 1981) have shown that 4-year-old aspen sprouts are killed as early as 9 months after infection, and that sapling mortality depends on time of infection (June infections did not result in mortality as often as July or September inoculations).

Source of Inoculum

Ascospores are presumed to be wind-borne (forcibly ejected from asci), while conidia are more likely to be spread by water, when they swell and extrude from beneath the bark.

Description

Cankers (Figs. 10a, b) average 2–5 cm in width, but expand along the long axis of the trunk or branch to lengths of up to 33 cm in as little as 4 years (rates measured after artificial inoculation of wounded aspen). However, discolored bark may extend beyond the actual canker for over 10 times that length (Hinds 1981). Smaller trees can be killed (usually due to sapwood decay rather than by girdling), and trunk (Libertella) decay occurs in larger trees.

The perithecial (sexual, *Cryptosphaeria*) state is most commonly encountered on year-old or older dead bark. It is less likely to be observed on very young trees or saplings, where *Cytospora* often fruits secondarily, confusing the diagnosis. Perithecia are produced in a pseudostroma, which consists of a slightly raised area composed of a mixture of fungal and bark tissue. Pseudostromata average 1–2 cm across but may extend up to 30 cm up and down the tree. Perithecia are about 0.4 mm in diameter, circular, black, and embedded in the pseudostroma, with black ostioles slightly protruding through the bark surface, giving it a "polka-dotted" appearance (Fig. 10c). Asci are long-stalked (Fig. 10d) and each ascus bears a small apical disc which does not turn blue in iodine, and contains eight ascospores. These ascospores (Fig. 10e) are yellow-brown, allantoid (sausage-shaped), 8–12 $\times$ 2–3 µm.

The asexual (conidial, *Libertella*) stage is one of the most frequently isolated fungi associated with a red heartwood stain and incipient decay of living aspen, but is not usually seen sporulating on field collections. It is commonly isolated in advance of decay columns produced by other fungi such as *Phellinus tremulae* (Basham 1958; Hinds 1981). When it does sporulate on its host, *Libertella* produces light orange acervuli which form raised pimples beneath the bark at the periphery of the canker. The acervuli bear whitish masses of filiform, nonseptate, strongly curved conidia, 15 $\times$ 1 µm.

Fig. 10. Cryptosphaeria canker of aspen and cottonwoods.

Figs. 10a, b. Long stem cankers on trembling aspen.

Fig. 10c. Dark punctate spots mark perithecial ostioles on cottonwood bark.

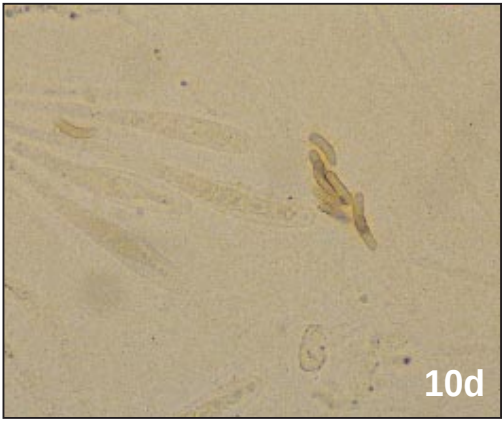

Fig. 10d. Immature asci. Photomicrograph (× 1000).

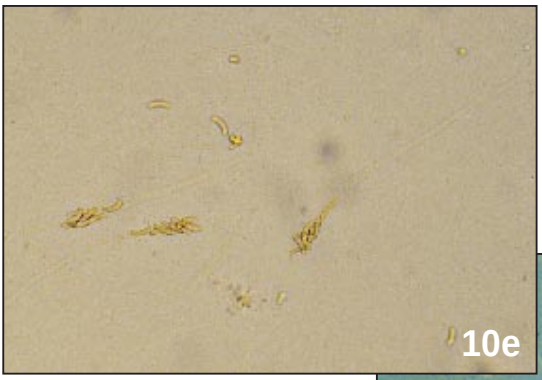

Fig. 10e. Long-stalked mature asci with brown ascospores. Photomicrograph (× 600).

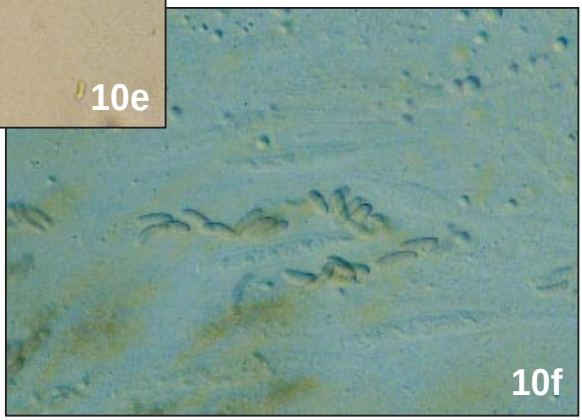

Fig. 10f. Mature ascospores. Photomicrograph (× 1000).

Key Diagnostic Features

- long, narrow cankers
- incipient and advanced, brown mottled trunk decay associated with a *Libertella* state in culture (1 cm^3 piece of stained wood surface-sterilized in 10% bleach, grown on malt agar)
- presence of many black perithecia in bark that has been dead for at least 1 year (*Cryptosphaeria* does not always produce perithecia on small trees. Confirmation by culture is necessary, especially because secondary infections and subsequent fruiting body formation by *Cytospora chrysosperma* may confuse the diagnosis.)

Impact

Although poorly documented in B.C., elsewhere *Cryptosphaeria* has frequently been isolated from redstain of aspen heartwood and incipient decay of aspen (Basham 1958). It is likely more common than current observations suggest, as secondary growth by *Cytospora* often obscures the primary infection.

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Hypoxylon Canker

Causal Agent

Entoleuca mammata (Wahlenberg:Fr.) J.D. Rogers and Y.-M. Ju. (Ascomycetes, Xylariariales)

Synonym = *Hypoxylon mammatum* (Wahlenberg) P. Karst. This older synonym is the name most commonly encountered in forest pathology literature. Rogers and Ju (1996), world authorities on *Hypoxylon*, believe that *E. mammata* is not a true *Hypoxylon* because of several distinctive features that will be described later.

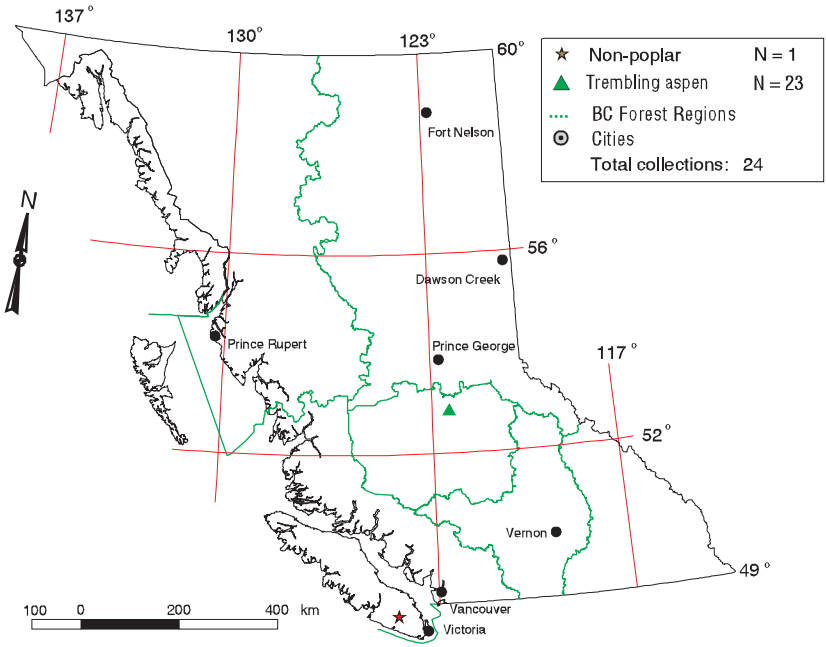
Hosts/Host Specificity

Hypoxylon cankers occur most commonly on *Populus tremuloides*, but are also rarely encountered on *Salix* spp. and *Alnus sinuata* (Regel) Rydb. (Sitka alder) in B.C. Hypoxylon canker has not been reported from other poplar species or hybrids in the province. Elsewhere in North America, it is common and damaging on various *Populus* species, especially *P. tremuloides* and *P. grandidentata* Mich., as well as *Alnus*, *Quercus*, *Malus*, *Betula*, and other hardwoods (Ju and Rogers 1996).

Time of Appearance

Cankers are perennial and thus may be detected any time of year. New cankers appear after stress or wounding.

Distribution of *Entoleuca mammata.*



Source of Inoculum

Infections are initiated from wind-borne ascospores, which are forcibly ejected from perithecia during periods of damp weather. *Entoleuca mammata* can live saprophytically on dead fallen trees, where it is associated with white rot of heartwood. Perithecia and ascospores may therefore be produced for several years after the death of the tree. Conidia, although prolific, are not considered to be an inoculum source, and are more likely to function as spermatia (sexual propagules) prior to the development of perithecia. Infections are more common on insect or sapsucker-damaged, drought-stressed, or otherwise stressed trees (Bélanger et al. 1989; Ostry and Anderson 1983).

Description

Cankers begin as yellow to orange, slightly sunken areas of bark on lower trunks or branches, and are often associated with insect damage, drought or winter damage. Cankers expand more rapidly longitudinally than horizontally, the surface becoming roughened and cracked, and a mycelial fan developing on the cambium layer (Figs. 11a, b). A few years after infection, cankered bark starts to separate from the underlying wood, as grey, dusty pillars of fungal tissue (hyphal pegs) 2–5 mm in diameter and 1–2 mm tall, covered with conidia, forces the bark outward (Fig. 11h). Conidia are hyaline, single-celled, 5.5–8.0 × 1.5–4.0 µm, and noninfective, although they occasionally germinate in culture. Perithecia develop in whitish to greyish crusts (Fig. 11c) on dark central areas of the canker, where the bark has split or sloughed off. Clusters of 10–30 perithecia may be fused into a single crust, or stroma, with the top of each perithecium nipple-like, 0.7–1.0 mm in diameter (Fig. 11d), opening with a darker central ostiole (Hiratsuka et al. 1995). When fresh dry stomata are sectioned, the mature hymenial layer is black and shiny inside. Asci are cylindrical, long-stalked, and contain eight ascospores (Fig. 11e) and a prominent rectangular apical plug which blues in iodine (Fig. 11f). Ascospores are blackish brown, elliptical, with one side flattened, 20–33 × 9–12 µm, with a straight germ slit appearing under high magnification as a pale line on the curved side of the spore (Fig. 11g).

Figs. 11a–h. Hypoxylon canker of trembling aspen, caused by *Entoleuca mammata*.

Figs. 11a, b. Typical cankers on trembling aspen.

Fig. 11c. Perithecia of *Entoleuca mammata* emerging through cracked bark on an aspen canker.

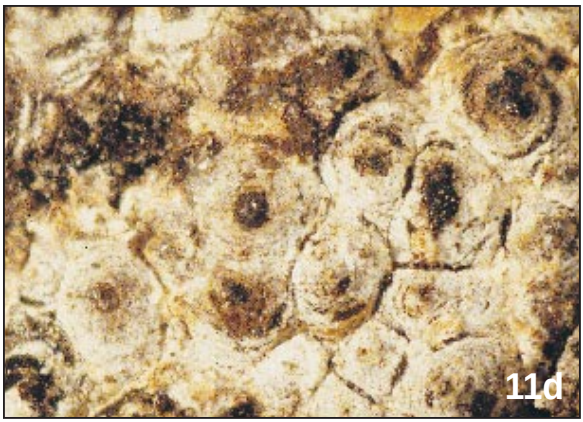


Fig. 11d. Perithecia (× 17).

Fig. 11e. Asci. Photomicrograph (× 250).

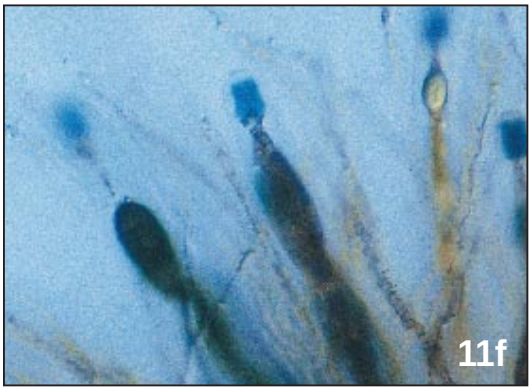


Fig. 11f. Ascus tip, stained blue with Melzer's reagent. Photomicrograph (× 600).

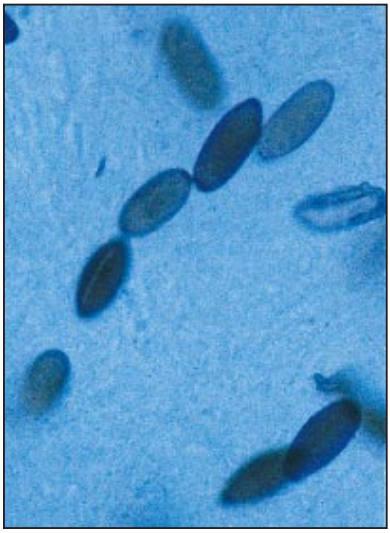


Fig. 11g. Ascospores, germ slit evident on concave side of two center spores. Photomicrograph (× 500).

Fig. 11h. Conidial columns pushing between bark and sapwood (× 15).

Fig. 11i. *Nemania serpens* stromata on trembling aspen. This fungus is common on dead decorticated poplars, and may be mistaken for *E. mammata*.

Figs. 11j–l. *Hypoxylon novemexicanum*, a saprophyte on fallen trembling aspen wood.

Fig. 11j. Stromata.

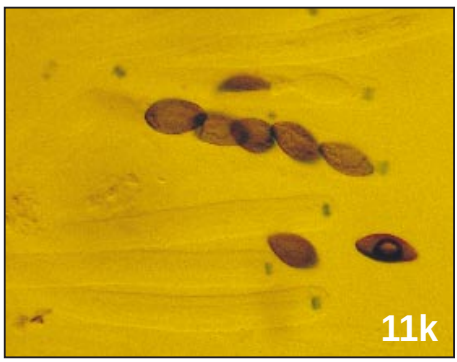


Fig. 11k. Small, flat ascus tip bluing in Melzer's reagent. Photomicrograph (× 300).

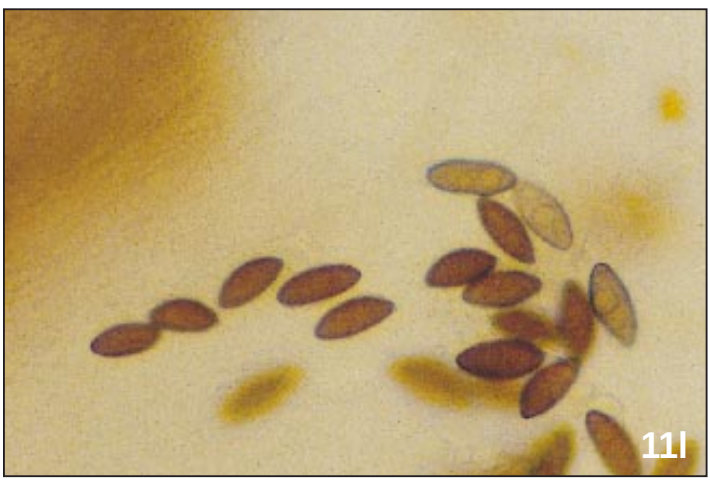


Fig. 11l. Ascospores, germ slit on convex side of spore. Photomicrograph (× 440).

Key Diagnostic Features

- cankers less than 0.5 m long
- cankers crusty, cracked
- grey perithecia
- grey dusty conidial pegs between bark and cambium

Look-alikes

Several species of *Hypoxylon* occur as saprophytes on aspen (see Host-fungus Index). Superficially, they may be confused with *E. mammata* on old, dead wood (Figs. 11i, j), but would not be encountered on fresh cankers on dying or recently dead trees. *Entoleuca mammata* fruiting bodies differ from *Hypoxylon* species in several ways (Ju and Rogers 1996), including:

- Stroma does not exude pigments when a small piece is placed in a 10% solution of potassium hydroxide (KOH) on a microscope slide or watch glass
- Ascospore germ slit (small pale line) occurs on the concave side of the spore. Ascospores of *Hypoxylon* species have germ slits on the convex side if the spores are inequilateral (Fig. 11l).
- Immature ascospores of *E. mammata* bear a cellular appendage. *Hypoxylon* species never have appendaged immature ascospores.
- The ascus tip contains a plug which turns blue in Melzer's reagent, is taller than broad, and is clearly visible at 400× under a compound microscope. *Hypoxylon* species have much reduced disc-like ascus apical structures that are much broader than tall, and are often hard to see even at high magnification (Fig. 11k).
- The asexual stage (anamorph) produces bark-rupturing hyphal pegs covered in conidia, unlike anamorphs of any *Hypoxylon* species.

Impact

In B.C., Hypoxylon canker has been infrequently collected, and has not been associated with heavy damage within stands. It has been collected from interior regions of the province (mostly in the Kamloops, Cariboo, and Prince George Regions) but has not been reported on aspen from coastal B.C. or on Vancouver Island. In eastern North America, Hypoxylon canker is a very important and damaging disease of aspen, especially in stands predisposed by weather or insect damage.

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Neofabraea Canker of Poplars

Causal Agent

Neofabraea populi G.E. Thompson (Ascomycetes, Helotiales) (anamorph = *Cryptosporiopsis* sp.). Other members of this genus have been transferred to *Pezicula* Tul. & C. Tul.

Hosts/Host Specificity

Neofabraea canker occurs on *P. tremuloides*, *P. trichocarpa*, and its hybrids in B.C. In Europe, it occurs also on *P. tremula* and hybrid aspen, and in Japan on *P. sieboldii*, *P. alba*, *P. tremula* var. *davidiana* × *canescens*, and *P. nigra* (Langhammer 1971).

Time of Appearance

Mature apothecia are produced in June, but there is no documented account of the disease cycle.

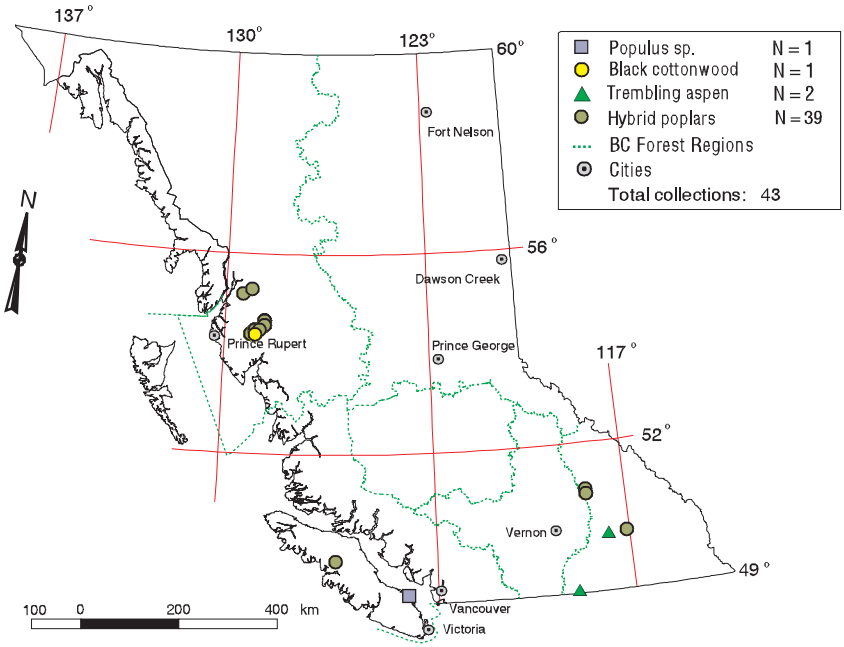
Source of Inoculum

Wind-borne ascospores appear to infect through lenticels (Roll-Hansen and Roll-Hansen 1969).

Description

Cankered areas first appear slightly more sunken than surrounding tissue, and bark may be discolored orange. Acervuli develop in concentric waves around the point of inoculation, usually around a lenticel; they first appear as pimple-like protrusions before they rupture the epidermis to expose their whitish to yellowish surface (Fig. 12a). Acervuli measure 0.5–1.5 mm in diameter and up to 50 µm thick, and are covered by hyaline, septate, simple or branched conidiophores measuring 25–35 × 4 µm. Conidia (Fig. 12d) are cylindrical to spindle-shaped, hyaline, nonseptate, and very large, 25–45 × 4–5 µm, often with granular cytoplasm (Thompson 1939).

Distribution of *Neofabraea populi*.



Apothecia develop in early summer, usually erumpent first near the center of the cankered area. They are pale brown to amber, fleshy, disc-shaped to cushion-shaped, and 0.5–2 mm in diameter (Fig. 12a). Asci (Figs. 12b, c) are borne on the surface of the apothecia, and are cylindrical to clavate, short-stalked, 80–112 × 9–12 µm, bearing eight ascospores. Ascospores (Fig. 12d) are oblong-ellipsoid, straight or slightly curved, or unilaterally flattened, hyaline, up to three septate, 16–22 × 5–6 µm (Funk 1981).

Key Diagnostic Features

- brownish to amber-colored, cushion-shaped apothecia
- erumpent whitish conidial stage
- large granular conidia

Look-alikes

This fungus may be superficially confused with Cytospora canker in the field but is easily distinguishable by its microscopic features.

Impact

Neofabraea may cause serious cankering in native and exotic poplars, especially hybrids and young nursery stock.

Fig. 12. Neofabraea canker.

Fig. 12a. Apothecia and acervuli on cankered poplar stem.

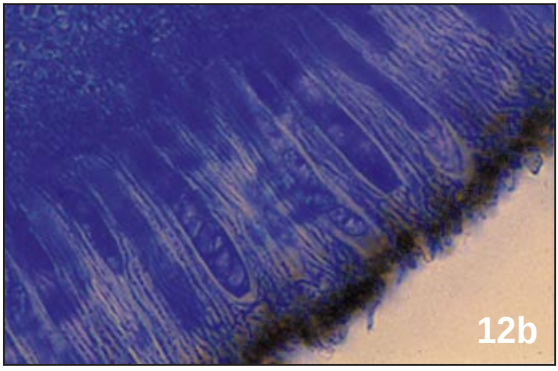


Fig. 12b. Immature ascus, ascospores. Photomicrograph (× 1000).

Fig. 12c. Thin section of apothecium. Photomicrograph (× 700).

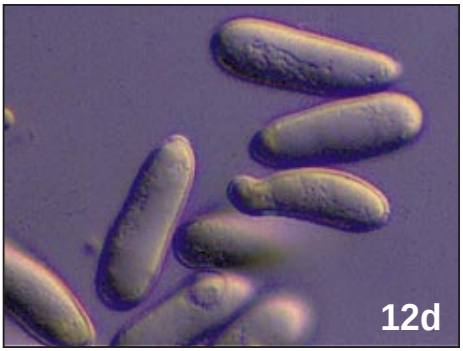


Fig. 12d. Conidia. Photomicrograph (× 1000).

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Bark Deformity Diseases of Poplars

Several species of fungi are found in consistent association with distinctively rough, swollen or corky poplar bark. In most cases, the bark deformity/fungus associations have not been confirmed by inoculation studies, and disease cycles are unknown. Some of the associated fungi may merely be using a niche created by other agents. Damage to yield and health of the tree is unknown, but the change in bark morphology following infection is striking enough to provoke disease diagnostic inquiries. Kaufert (1937) proposed that certain symptoms, such as cork-bark, might be initiated by a physical injury, which induces wound periderm development, which in turn provides an entrance court for secondary fungi. However, the actual infection process of these fungi has not been documented.

Various distinctive types of bark deformities and swellings, along with their associated fungi, follow.

Rough-bark Diseases of Aspen

Causal Agent

Rhytidiella baranyayi Funk & Zalasky (Ascomycetes, Cucurbitariaceae)

This fungus is associated with corky rough bark of aspen throughout B.C. (Funk and Zalasky 1975). The corky areas begin as swollen, diamond-shaped proliferations (Figs. 13c, d) which eventually split in the center (Fig.13a). In time, affected bark becomes dark, roughened, and longitudinally ridged, extending almost the full length of the trunk on heavily infected trees (Fig. 13b).

Ascocarps are erumpent from the corky periderm, black, globose, 175–250 µm in diameter (Fig. 13e), opening with an apical pore to release cylindrical, bitunicate asci, 35–65 × 8–15 µm (Fig. 13f), containing eight hyaline, curved three-septate ascospores, 15–25 × 2–4 µm (Fig. 13g). This species has no known conidial state in nature, although hyaline, one-septate, worm-like conidia, 10–14 × 2–2.5 µm, are produced in culture.

A second fungus is commonly associated with rough bark disease of aspen: *Lahmia kunzei* Korber (= *Parkerella populi* Funk) (Ascomycetes, Lahmiales). This species is found on *P. tremuloides* stems in North America, and in Europe it is encountered on *P. tremula*, *P. alba*, and other hardwoods (Eriksson 1986). In B.C. the fungus is most common in the central interior (Quesnel, Williams Lake). It was first identified by Funk (1976) who described it as a new genus, *Parkerella*. Ten years later, Ericksson (1986) realized that *Parkerella populi* was synonymous with *Lahmia kunzei*, a species originally described in Europe on *P. tremula* in 1861.

Fruiting bodies of *L. kunzei* are consistently associated with rough bark in fissures on aspen in B.C., although they are not believed to cause the roughening. This fungus is never found on smooth bark or on exposed wood, but is occasionally found on bark roughened by agents other than *R. baranyayi*. The small, black ascomata are erumpent from the bark, 125–200 µm in diameter × 240–250 µm tall, breaking open at the top with irregular fissures to release long-stalked, clavate, J-asci bearing eight, hyaline, crescent-shaped, one-to three-septate ascospores measuring 18–33 × 4 µm.

Lahmia kunzei was the only fungus observed on deeply fissured, black bark of declining triploid *P. tremuloides* × *P. tremula* hybrids in Wisconsin (Ostry 1986), but this fungus was not proved to cause the decline.

The surface of aspen bark may also be roughened up to 1.2 m from the ground from vole feeding, which takes place under snow during the winter (Hinds and Krebill 1975).

Fig. 13. Bark disorders of aspen.

Fig. 13a. Rough-bark of aspen, developing in longitudinal patches on the trunk.

Fig. 13b. Aspen with entire trunk affected by cork-bark disease.

Figs. 13c, d. Small areas of cork-bark (Fig. c), showing depth of affected tissue in Fig. d.

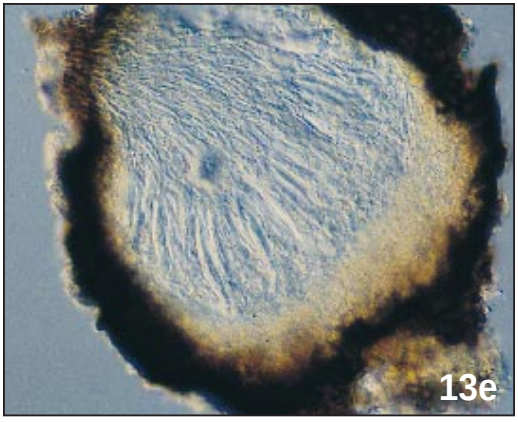
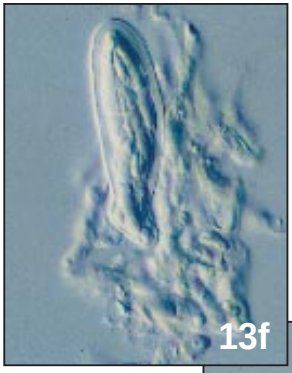
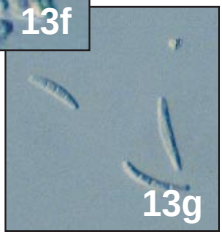


Fig. 13e. Perithecium (ascocarp) of *Rhytidiella baranyayi*. Photomicrograph (× 300).





Figs. 13 f, g. Ascus (Fig. f) and ascospores (Fig. g) of *R. baranyayi*. Photomicrographs (× 750).

Fig. 13h. Diplodia bark swellings associated with *Seimatosporium etheridgei* on aspen.

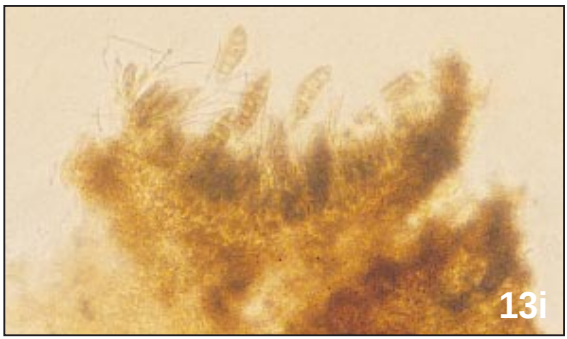


Fig. 13i. Conidia from acervulus of *S. etheridgei*. Photomicrograph (× 220).

Fig. 13j. Diplodia galls on cottonwood branches.

Fig. 13k. Diplodia gall on cottonwood.

Aspen Stem Galls and Bark Proliferation

Causal Agent

Diplodia tumefaciens (Shear) Zalasky (teleomorph = *Keissleriella emergens* (P. Karst.) Bose) (Ascomycetes, Lophiostomataceae.)

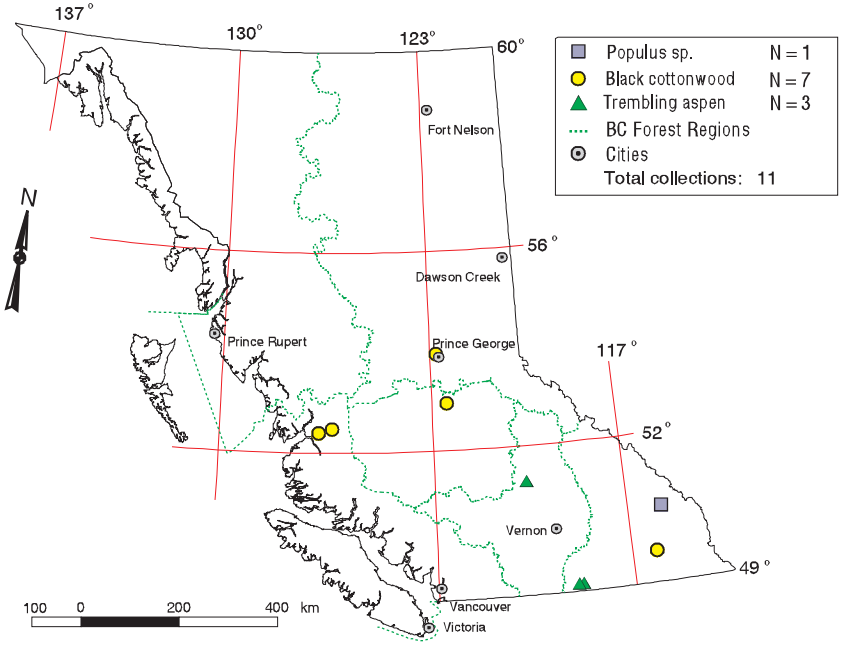
This fungus has been shown to cause oval to globose branch galls (Figs. 13j, k) and rounded, tumorous patches of bark proliferation via inoculation experiments (Zalasky 1964a).

In B.C., *D. tumefaciens* is associated the three native species: *P. tremuloides*, *P. trichocarpa*, and *P. balsamifera*, but branch galls are more common on the last two species. It has been reported on Lombardy poplar and *P. deltoides* hybrids in the prairie provinces (Zalasky 1965). The fungus is also associated with similar symptoms on *P. tremula* in Europe (Zalasky 1964b). Large galls may persist on infected trees for over 20 years (Zalasky 1965). Stem defects are caused by swellings and suppressed growth (Zalasky 1964a; Zalasky et al. 1968).

Pycnidia on galled bark are a diagnostic feature of this fungus. They are black, globose, erumpent from infected bark, 250–450 µm in diameter with an apical ostiole. Conidia are oblong to irregularly ellipsoid, 28–40 × 9–15 µm, hyaline to yellowish, single-celled to rarely one or two septate. Occasionally cylindrical microconidia measuring 6–12 × 2–3 µm are also present in the pycnidia.

The sexual state, *K. emergens*, is produced in summer on roots that are infected by conidia produced in the galls. Diseased roots are irregularly warted and swollen, becoming covered with a black stroma on the upper surfaces, which bears masses of black pseudothecia 180–225 × 225–380 µm. These fruiting bodies are conical to globose, with an apical ostiole lined with setae up to 180 µm long. Asci are bitunicate, clavate, 65–85 × 9–12 µm, and bear eight fusoid hyaline ascospores measuring 20–27 × 4–5 µm, which are constricted in the middle and become pale brown and one to three septate when mature (Zalasky 1974).

Distribution of *Diplodia tumefaciens.*



Leciographa gallicola Funk (Ascomycetes, Patellariaceae) (anamorph = *Seimatosporium etheridgei* Funk) is frequently associated with Diplodia branch galls and bark proliferation (Funk 1979, 1985). However, the anamorph of this fungus, *S. etheridgei*, was originally described separately (Funk 1978) as the putative cause of bark proliferation on aspen stems. Not until several years later did Funk (1985) link the anamorph and teleomorph. The primary cause of both the stem galls and the bark proliferations (Fig. 13h) is now believed to be *D. tumefaciens* with *L. gallicola* invading secondarily once the abnormal growth has developed. No branch galls have been found that contain *Leciographa* without *Diplodia*, although this is often the case with bark proliferations on stems. The presence of *Leciographa* in the stem might suppress sporulation of *Diplodia*, thus giving the appearance that the former was the primary pathogen (Funk 1985).

The *Leciographa* state produces black apothecia on the surface of galled and roughened bark. Apothecia are solitary or clustered, flat, disc-like, 0.5–0.9 mm in diameter with tissues bluing in Melzer's reagent. Asci are clavate, bitunicate, 85–100 × 10–18 µm, bluing entirely in Melzer's reagent, bearing eight ellipsoid dark brown ascospores 27–50 × 8–12 µm. Ascospores are three to seven septate with the end cells paler. The *Seimatosporium* state produces conidia in shallow irregular black acervuli 0.1–0.4 mm in diameter on bark. Conidia are similar in appearance to ascospores, three to six septate, 30–44 × 13–15 µm (Fig. 13i).

Cucurbitaria staphula (Dearn.) R.H. Arnold & Russell (Ascomycetes, Dothideales) (anamorph = *Pseudodichomera*) also commonly sporulates on Diplodia galls, and is recognizable by black stromata containing locules filled with bitunicate asci measuring 144–240 × 18–25 µm, bearing eight muriform ovate ascospores. Ascospores are brown, three to seven septate, 27–48 × 12–16 µm. The conidial state is similar in appearance, with a loculate stroma bearing brown muriform conidia measuring 12–16 × 8–10 µm.

Cork-bark of Cottonwood

Causal Agent

Rhytidiella moriformis Zalasky

A second species of *Rhytidiella*, *R. moriformis*, is associated with *P. balsamifera*, causing it to produce the furrowed bark ridging so common that it is often mistaken for the natural characteristic of the tree (Zalasky 1968). *Rhytidiella moriformis* produces erumpent perithecia on the roughened bark. The contents of the perithecia stain red in Melzer's reagent. The ascospores are larger than those of *R. baranyayi*, and are hyaline to yellowish or olive-brown, sinuous, 11–15 septate when mature, 60–100 × 3–6 µm. Small, 115–250 × 75–100 µm, flattened to conical ostiolate pycnidia are also present on the bark, bearing up to 10 septate hyaline to olivaceous conidia measuring 45–116 × 2.5–6 µm (Zalasky 1968).

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Foliar and shoot diseases are characterized by discolored spots, or blackening and shriveling of whole leaves and succulent current-year growth. Occasionally, interactions between a parasitic fungus and its hosts results in deformed growth (blisters, hypertrophy) of foliar tissue. The life cycles of foliar fungi are often finely choreographed to the spring leaf development of their hosts. The overwintering spore stages of many different foliar blight fungi (usually in fallen leaves on the ground) mature in the spring, just as the buds flush. At this time spores usually become airborne, and infect young leaves while their cuticles are still soft and they have not yet fully expanded. Spring is thus the most likely time for a tree to become infected with foliar fungi, and spring weather that is unusually mild and wet tends to exacerbate infection levels and subsequent disease development. Foliage also tends to be susceptible to opportunistic blight fungi after periods of stress, or late in the growing season when tissues begin to senesce.

Host tissues parasitized by fungi may be rapidly overgrown by secondary saprophytes which obscure symptoms and signs of the primary cause of the disease. Saprophytes also greatly reduce the likelihood of obtaining a pure culture and correctly identifying the causal agent of the disease unless highly specialized selective media are used. When fungi are not sporulating on obviously diseased foliage, cultures should be initiated as soon as possible from material excised from the zone between healthy and diseased tissue, where the pathogen is most active. This technique will help to avoid contamination from surface fungi and bacteria. A leaf portion may be surface-sterilized with a 10% aqueous solution of household bleach and then rinsed several times with sterile distilled water, prior to culturing on 2% water agar.

Fruiting bodies associated with leaf spots might be immature at the time of collection. These fungi might not fruit until the host tissue is dead or nearly dead. The causal agent may be cultured using the techniques previously described, or, if fruiting bodies are very close to sporulation, the following inexpensive shortcut may be used. Place the leaf, fruit or stem segment in a plastic bag, inflate the bag (humidity from one's breath usually provides sufficient moisture; addition of more water or damp paper towels is usually excessive), and incubate for 1–3 days, checking for sporulation daily. The fruiting bodies might be overcome by aggressive saprophytes, but if the sample is not contaminated by soil or damaged by insects, this method often works well.

Certain groups of plant parasites, such as rusts, have alternate hosts, and may not be reliably identified to the species level from spore stages produced on one of the hosts. As many spore stages as possible should be collected, by sampling from tissues of all ages and degrees of infection.

In temperate regions such as B.C., fruiting bodies of certain groups of fungi, in particular those causing foliar diseases, require overwintering prior to maturation of the sexual state. Symptoms and non-sporulating fruiting bodies will be evident on diseased plants collected during the current growing season. To confirm the identity of the fungus, it might be necessary to collect sporulating fruting bodies on fallen overwintered foliage in the spring.

Key to Common Foliar Diseases and Shoot Blights

1. Fungus signs such as tissue,* fruiting bodies (black spots), or spore masses (white, orange, pink, green) visible either by the unaided eye or by using a hand lens or dissecting microscope 10
1. Fungus signs as previously described not evident on diseased leaves but fungus tissue may be visible in material examined using a compound microscope 2

2. Wounds, holes, or excess sap exudates (honeydew) present 3
2. Wounds, holes, or excess sap absent 4

3. Holes or puncture wounds in leaves, insect frass, or honeydew present, or larvae or aphids present inside deformed or damaged host tissue . . . . . .insect damage
3. Holes ("shot-holes") in browned aspen leaves but no insect frass or insect activity; adjacent dead or fallen leaves bearing black "ink spots;" petioles often remaining green . . . . . .*Ciborinia whetzelii*

4. Hypertrophied (often with strange outgrowths), stunted, twisted, or malformed leaves or petioles present 5
4. No hypertrophied growths, stunting, or malformation but foliage may be discolored or withered 9

5. Malformed areas discrete; limited to strange, often brightly pigmented outgrowths on all or part of the leaf 6
5. Malformed areas generally more systemic, often involving entire buds or shoots 7

6. Malformed areas cup-like, several millimeters to several centimeters across (but thickness relatively unchanged) on the leaf blade; bright yellow early in the growing season, often darkening to orange or brown by late summer, with fungal tissue present on surface of cup . . . . . .*Taphrina* spp.
6. Malformed areas not cup-like, instead covered with red or orange finger-like or furry growths that are thicker and tougher than unaffected tissue; no fungal tissue present . . . . . .mite damage

7. Buds swollen, covered in resin and not flushing; no aphids present in tissues . . . . . .bud midge damage
7. Buds not swollen, leaves and shoots stunted, chlorotic, or necrotic (brown-flecked or twisted and withered) 8

8. Leaves, petioles, or entire shoots flecked, discolored, twisted, necrotic, and/or stunted but no biotic agents evident . . . . . .chemical damage
8. Leaves, petioles, or entire shoots suddenly withering and turning black; most severe on exposed side and outer branches of tree; no fungal tissue present . . . . . .frost damage

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* To test for the presence of fungal tissue, section leaf with a straight-edge razor, stain with lactophenol/cotton blue, and examine microscopically. Fungal tissue will differentially absorb the blue stain. If the fungus cannot be identified based on host symptoms alone, culturing of infected tissue might be required for diagnosis. Fruiting body production can be encouraged by incubating the infected leaf in an inflated plastic bag. It is usually not necessary to add moistened paper towels to the bag unless the leaf is desiccated. Excess moisture encourages growth of saprophytic bacteria and fungi.

9. Small (about 5 mm in diameter) circular discolored spots on cottonwood (if microscopic examination reveals a fungus is sporulating on spots, see also description of *Phaeoramularia maculicola*) . . . . . .leaf gall midge
9. Larger areas affected; undersurface of leaf turning bronze along margins and major veins; in some cases entire abaxial surface bronzed or even blackened while upper surface remains green; on cottonwoods and their hybrids . . . . . .leaf-bronzing mite
(refer to section on *Arthropod Damage Often Confused with Poplar Diseases*)

10. Black withered leaf spots and shoots ("shepherd's crooks") scattered throughout the tree but often more pronounced in the lower canopy and on understorey trees, and with dark olive-green conidial deposits present on dead tissues . . . . . .Venturia blight
(*V. macularis* – aspen; *V. borealis* – aspen, but leaf spots only, and in northern regions of the province; *V. populina* – cottonwoods and hybrids)
10. Not as in previous description (diseased areas appearing as blotches or spots on affected leaves) 11

11. Diseased areas consisting of large necrotic blotches covering from 25% to the entire leaf area; fruiting bodies black, not sporulating while leaves still attached, and clearly visible to the unaided eye (> 0.3 mm in diameter) 12
11. Diseased areas or fruiting bodies smaller or different color or producing spores 14

12. One or a few large black thick circular "ink spots" up to 1 cm in diameter, consisting of non-sporulating tissue embedded in the brown portions of aspen leaves . . . . . .*Ciborinia whetzelii*
12. Numerous (often hundreds) of small (< 1 mm in diameter) black fruiting bodies forming in the browned foliar tissue of various poplars other than aspen 13

13. Margins of infections always black, with a feathery border similar to ink dropped on blotting paper, fruiting bodies angular (about 0.5 mm in diameter); common on Vancouver Island and the west coast of B.C. . . . . *Linospora tetraspora*
13. Margins of infections at first yellow, then brown, entire; fruiting bodies smaller (about 0.2 mm in diameter) round; known only from the Prince Rupert Region . . . . . .*Guignardia sp. nov.*

14. Orange powdery pustules of fungus spores appearing on green leaves in early summer, becoming cushion-like, waxy and brown on dead or dying leaves at the end of the growing season . . . . . .*Melampsora* spp. (*M. medusae* on aspen, *M. medusae f. sp. deltoidae* on cottonwood hybrids on the Lower Mainland and Vancouver Island; *M. occidentalis* on native cottonwoods and some hybrids)
14. Fruiting bodies or spore deposits not orange and powdery 15

15. White fluffy or powdery fungal growth on the upper surface of chlorotic, senescent or stressed leaves . . . . . .powdery mildew
15. No white superficial fungal growth; necrotic spots usually 1–2 mm or less in diameter; on green or chlorotic leaves; spots sometimes coalescing to form larger areas 16

    16. Leaf spots frequently coalescing to form larger rounded necrotic spots, which bear black pinprick-like pycnidia (visible using a hand lens) in the centers . . . . . .*Septoria populicola*
    16. Leaf spots rarely coalescing; freckle-like; no pycnidia present but many conidia forming in scab-like mounds and breaking through the epidermis in spots . . . . . .*Marssonina* spp. (*M. populi* on native cottonwoods; *M. brunnea f. sp. trepidae* on aspen; *M. brunnea f. sp. brunnea* on hybrid poplars on Vancouver Island and the lower mainland)

Ink Spot Disease of Aspen

Causal Agent

Ciborinia whetzelii (Seaver) Seaver (Ascomycetes, Helotiales)

Hosts/Host Specificity

In B.C., *C. whetzelii* is widespread on trembling aspen, and is the only species of *Ciborinia* on poplar. It is most frequently encountered in the interior of the province. The sexual state, although likely to occur here, has not been documented in B.C. Found throughout Canada and the northern United States on trembling aspen, *C. whetzelii* also occurs rarely on balsam poplar (Baranyay and Hiratsuka 1967).

Time of Appearance

Brown foliar discoloration becomes evident approximately one month after infection, which occurs in the spring while new leaves are expanding.

Source of Inoculum

Overwintered sclerotia germinate on the surface of deep moist duff under aspen, and become covered with hundreds of small pale brown apothecia. These apothecia forcibly eject the ascospores which become wind-borne in the spring and infect expanding new leaves.

Description

Heavy infestations result in patches of dead, browned leaves which are visible in aerial surveys. Frequently, the petioles of infected leaves remain green well after the remainder of the leaf has turned completely brown (Fig. 14a). Browning of foliage is followed in approximately one month by development of circular to elliptical black sclerotia on the infected leaf blades. Sclerotia (Fig. 14b) are 2–8 mm in diameter, black, circular to oval or with rounded irregular outlines, with a distinctive palisade layer of fungal cells just under the black outer surface (Baranyay and Hiratsuka 1967). Sclerotia begin to fall from infected leaves as early as mid-July. The infected leaves may remain attached, bearing characteristic "shot-holes" until the fall. The sexual (apothecial) state of the fungus has not been found in B.C. but it has probably been overlooked because it occurs in early spring, and pathology surveys usually take place in the summer and fall when symptoms are evident on the tree. Apothecia are produced by the hundreds on the surface of fallen overwintered sclerotia. They are small, 2–10 mm in diameter, stalked and cup-shaped, with stalks 5–25 mm long. Asci are club-shaped, 160–180 × 11–12 µm, containing eight ascospores and a small apical plug that stains blue in Melzer's reagent. Ascospores are single-celled, ovoid, hyaline, 7–10 × 3–4 µm.

Look-alikes

Ciborinia seaveri Groves & Bowerman produces symptoms similar to *C. whetzelii*, but the sclerotia are smaller in size averaging 3–5 mm in diameter, and occur mainly on veins and petioles rather than the leaf blades. *Ciborinia seaveri* has not been reported in B.C., but is common in Alberta and eastern Canada (Baranyay and Hiratsuka 1967).

Key Diagnostic Features

- random clusters and patches of browned aspen leaves, often with petioles remaining green
- black sclerotia or "shot-holes" on browned aspen leaves

Impact

Aspen ink spot occasionally causes early defoliation of aspen, with outbreaks reported as early as 1960 (Molnar 1964). Historically, heavy outbreaks have occurred in northern B.C. from

Mile 400–430 of the Alaska Highway (Molnar 1964; Molnar et al. 1969); near Osprey Lake in the Kamloops District; and along the B.C.–U.S.A. border in the Nelson District (Molnar et al. 1965).

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Fig. 14. Ink spot disease of aspen.

Figs. 14a, b. Sclerotia of *Ciborinia whetzelii* on browned aspen foliage.

Linospora Leaf Blight

Causal Agent

Linospora tetraspora G.E. Thompson (Ascomycetes, Diaporthales)

Hosts/Host Specificity

Black cottonwood is the most common native host in B.C. Susceptible hybrid crosses include T×D, T×N hybrids, and T×M hybrids (rarely, at low levels). Aspen is not affected.

Linospora tetraspora was originally described in 1939, based on collections made in Ontario on *Populus balsamifera* (as *P. tacamahaca)*. Thompson (1939) noted that the blight was widespread on *Populus balsamifera* in Canada from Quebec to British Columbia. Barr (1978) described a similar distribution range for this fungus, with the addition of an United States record from Vermont. Based upon published records, *Linospora tetraspora* appears to be host-specific to *Populus* sections *Tacamahaca* and *Aigeiros* (Callan and Ring 1994) in North America. There are no published records of this fungus on other continents.

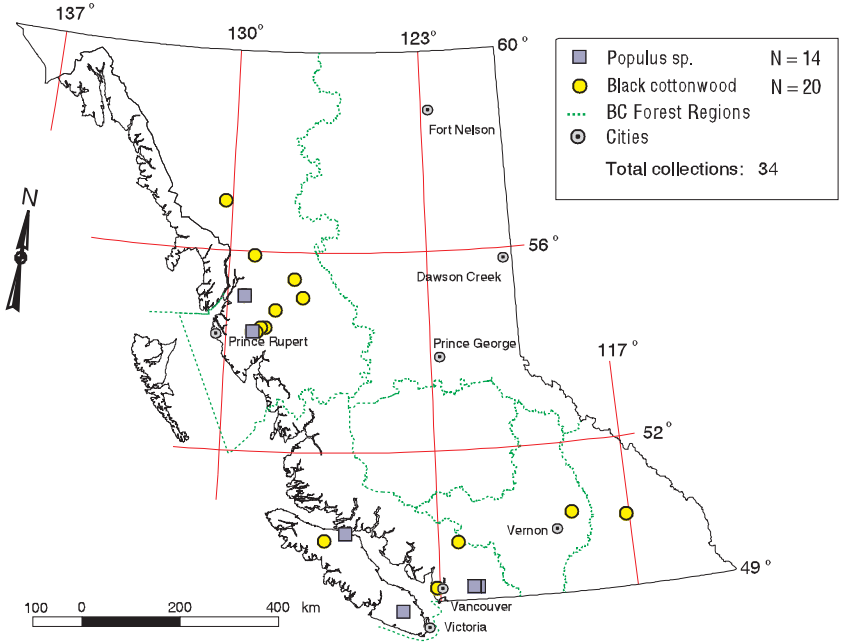
Time of Appearance

First signs of leaf spots start to appear in late May to early June. Diagnostic symptoms (black blotches, feathered edges) appear in late June, when fruiting bodies first start to develop.

Source of Inoculum

Newly flushed leaves are infected from inoculum on overwintered dead leaves on the duff surface. The black spots (pseudostromata) on fallen dead leaves contain perithecia which eject ascospores during periods of damp weather in the spring. Airborne ascospores are the only known source of inoculum for the single annual cycle of the disease. A microconidial stage is present in the fall, but microconidia are not thought to be infective, and are probably involved in sexual recombination of the fungus.

Distribution of *Linospora tetraspora*.



Description

Early-season infections first appear as black to dark brown discolored blotches on leaves. The dark stain advances more quickly along the leaf veins, creating the appearance of ink dropped on blotting paper (Figs. 15b, c). Eventually, entire leaves become completely blighted (Fig. 15e). The centers of the darkened blotches on the leaves lighten to tan by mid to late summer (Fig. 15d), and become evenly spotted with black, rectangular spots about 1 mm × 0.5 mm (Fig. 15f). These spots consist of a shield or clypeus composed of mixed host and fungal tissue covering a single embedded perithecium which opens to the upper leaf surface with a laterally beaked ostiole. Perithecia mature in the spring on fallen overwintered leaves (Fig. 15g). Asci are cylindrical, 175–230 × 6–9 µm, and contain only four spores (Fig. 15h). Ascospores are filiform, hyaline, straight or curved and twisted around each other while still in the ascus, 5–8 septate, 155–200 × 1–2 µm (Fig. 15i). Prior to perithecial development, a *Melasmia*-like conidial state produces hyaline, globose, conidia 2.5–3.0 µm in diameter. The function of these conidia is not known; they do not appear to be infective, but may act as spermatia.

Key Diagnostic Features

- large black "ink blotch" spots on otherwise green leaves midsummer
- centers of ink blotch spots turn tan by mid to late summer, and become dotted with black rectangular fruiting bodies about 1 mm × 0.5 mm; entire leaves are often affected
- persistent tan-colored dead leaves in the lower crown, which have a "leopard spot" appearance due to hundreds of regularly spaced fruiting bodies.

Look-alikes

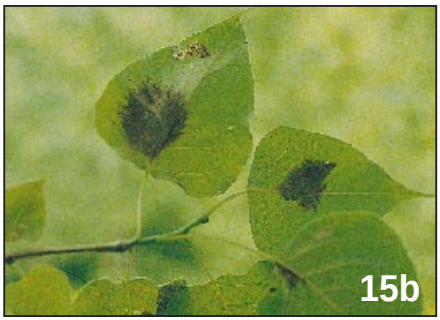
An undescribed species of *Guignardia* produces similar symptoms, but fruiting bodies are smaller, and lesions are limited to cottonwood in the Prince Rupert Region (B. Callan, unpublished observations).

Impact

This disease can severely defoliate native cottonwood in coastal plantations (Fig. 15a), and limit the productivity of certain hybrid poplars in years with prolonged damp springs.

Fig. 15. *Linospora* leaf blight of cottonwoods and hybrid poplars.

Fig. 15a. Heavily blighted T×D trees in a Fraser Valley plantation.



Figs. 15b, c. Characteristic “ink spot” appearance of infections during the growing season.

Fig. 15d. Blighted spots on leaves turn tan as the infection progresses.

Fig. 15e. Developing *L. tetraspora* ascostromata on completely blighted leaves.

Figs. 15f, g. Macroscopic view of developing (Fig. f) and mature, overwintered (Fig. g) ascostromata (× 5; × 10).

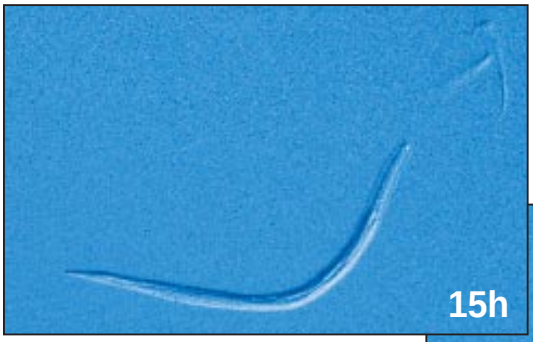


Fig. 15h. Ascus. Photomicrograph (× 500).

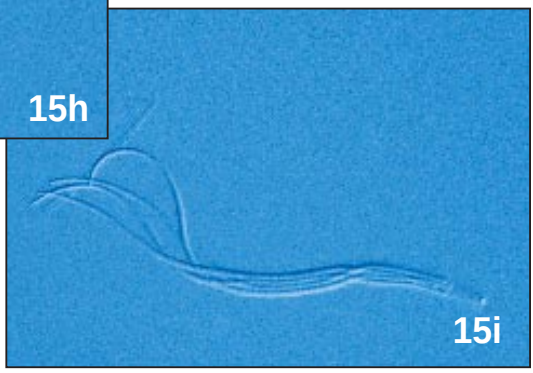


Fig. 15i. Broken ascus, with emerging filiform ascospores. Photomicrograph (× 600).

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Marssonina Leaf Blights

Causal Agents

Marssonina brunnea (Ellis & Everh.) Magnus *f. sp. trepidae* Spiers (teleomorph = *Drepanopeziza punctiformis* Gremmen) – (Ascomycetes, Helotiales)

Marssonina brunnea (Ellis & Everh.) Magnus *f. sp. brunnea* Spiers

Marssonina castagnei (Desmaz. & Mont.) Magnus. (teleomorph = *D. populi-albae* Kleb.)

Marssonina populi (Lib.) Magnus (teleomorph = *D. populorum* (Desmaz.) Höhn.).

Hosts/Host Specificity

Marssonina brunnea f. sp. trepidae is pathogenic to *P. tremuloides* in B.C. and elsewhere in North America. In Europe it is pathogenic to *P. tremula*.

Marssonina brunnea f. sp. brunnea was reported for the first time in the Pacific Northwest in 1996, on hybrid (T×D) poplar clones. Its identity was confirmed by conidial measurements and by leaf disc assays. The latter proved it was not pathogenic to local *P. tremuloides* (Newcombe and Callan 1996). Elsewhere in North America it is pathogenic to *P. deltoides* and its hybrids with *P. nigra* (Newcombe 1996).

Marrssonina castagnei occurs on *P. alba* in B.C.

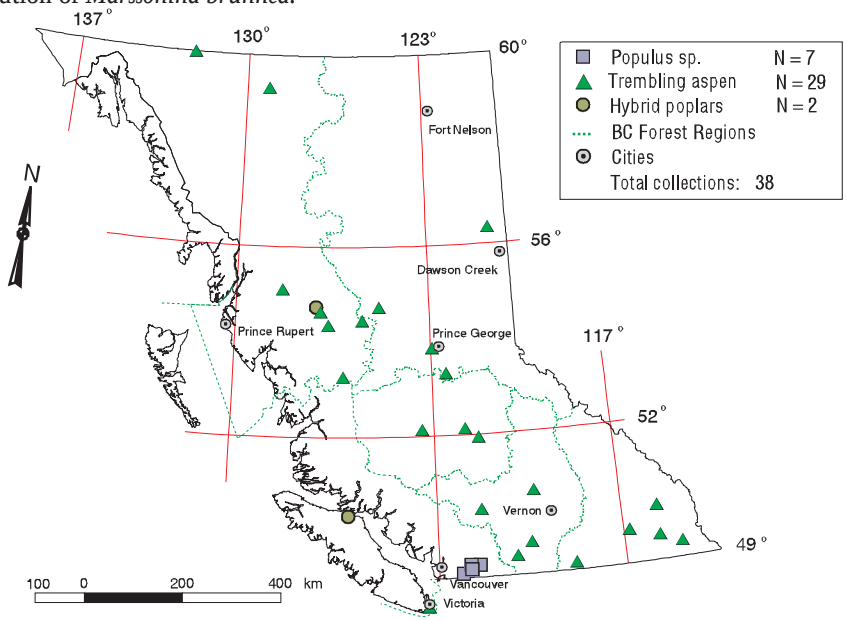
Marrssonina populi occurs on *P. trichocarpa*, T×N hybrids, and *P. nigra var. italica* in B.C., and elsewhere is specific to *Populus* sections Aigeiros and Tacamahaca (Newcombe 1996).

P. maximowiczii and its hybrids appear to be resistant to Marssonina blights.

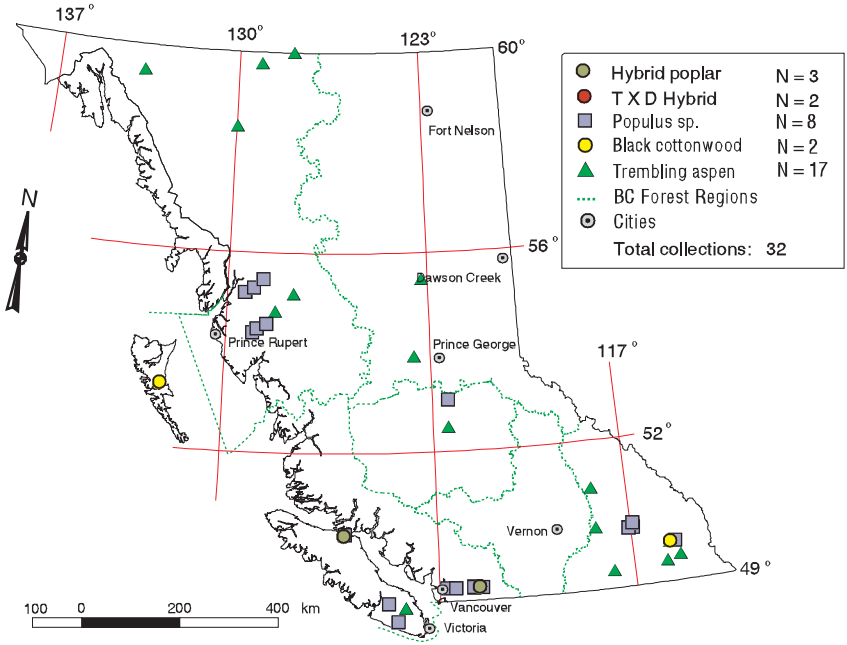
Time of Appearance

Small, angular leaf spots first appear in spring and early summer, while stem lesions, especially in damp crowded conditions in stool beds, may persist longer than one growing season. Cellerino et al. (1978) have shown that there is a correlation between leaf age and susceptibility to *M. brunnea*. Leaves have relatively low susceptibility during flushing, with increased susceptibility during expansion. Susceptibility decreased in mature leaves, and increased again when leaves became senescent.

Distribution of *Marssonina brunnea.*



Distribution of *Marssonina populi*.



Source of Inoculum

Infection of leaves and shoots in the spring is caused by conidia and possibly ascospores that are produced in infected fallen leaves and in shoot lesions on the tree. The sexual (*Drepanopeziza*) stages of *Marssonina* species are poorly documented in B.C. so their role in spreading disease is unclear. Secondary spread of conidia during the growing season is by wind and rain during wet weather. There is also evidence (Spiers and Wenham 1983) that *M. brunnea* is transmitted via infected seed capsules.

Description

The small angular leaf spots on the various poplar hosts range from 1 to 5 mm across, and may coalesce to form larger necrotic areas. Spots produced by *M. brunnea f. sp. brunnea* (Fig. 16f) tend to be on the small side of this range, and those produced by *M. populi* tend to be dark (Fig. 16i), and on the large side of this range, coalescing into vein-delimited blotches on some of the T×D hosts (Fig. 16j) (Spiers 1984). Acervuli range from 0.2 to 0.4 mm in diameter, and the developing mass of orange conidia erupts from beneath the epidermis, forming rough scabby areas. Occasionally *M. brunnea f. sp. brunnea* forms stem lesions on T×D hybrid poplars in stool beds in B.C. (Figs. 16d, e).

Microscopic features of *Marssonina* species found in B.C.:

Conidial measurements (Spiers 1988) are from lactoglycerol (*see Field and Laboratory Techniques section for formulation*). Measurements from lactoglycerol and water should not be compared because spore sizes tend to be larger if they are mounted in water.

Figs. 16a–c. Leaf blight of aspen caused by *Marssonina brunnea f. sp. trepidae*.

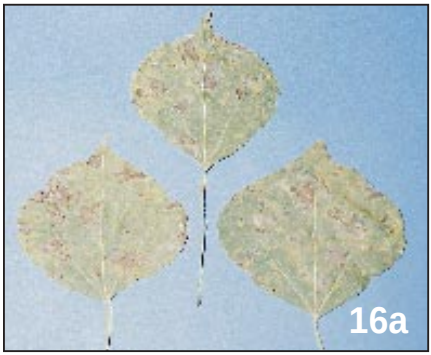
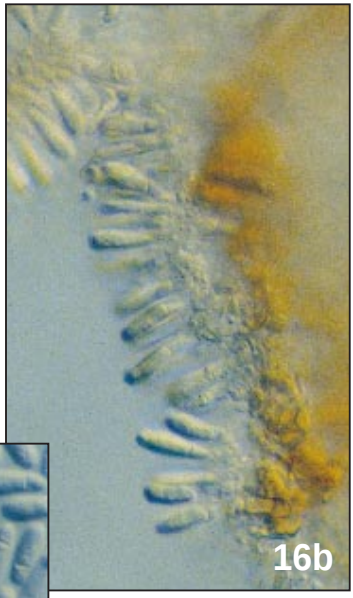
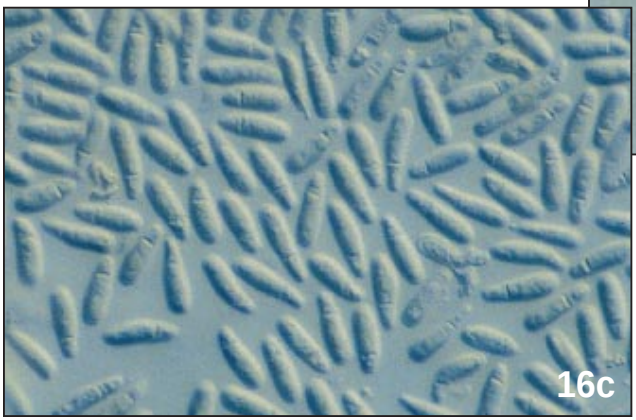


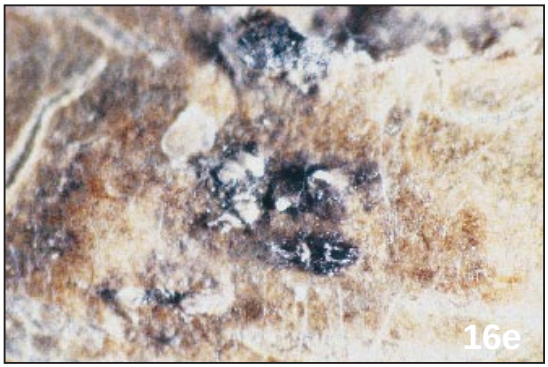
Fig. 16a. Leaf spots on *P. tremuloides*.





Figs. 16b, c. Conidia and conidiophores scraped from a *P. tremuloides* leaf surface. Photomicrographs (× 450; × 700).

Figs. 16d–h. Leaf and stem blight of hybrid poplars caused by *M. brunnea f. sp. brunnea*.



Figs. 16d, e. Stem lesions on a T×D hybrid poplar (Fig. 16d) planted in a stool bed, with pale conidial tendrils evident in Fig. e (× 15).

Fig. 16f. Profuse small spots on foliage of hybrid poplar.

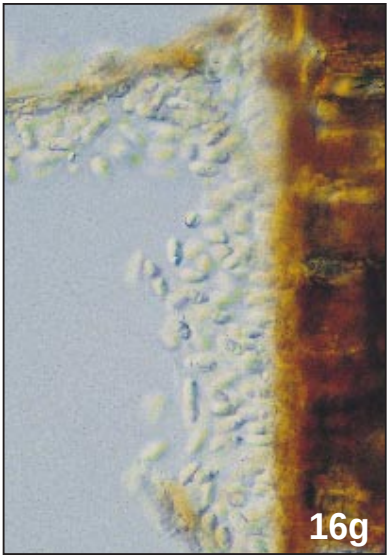


Fig. 16g. Section through a T×D leaf spot, showing epidermis raised above the conidiophores and conidia. Photomicrograph (× 500).

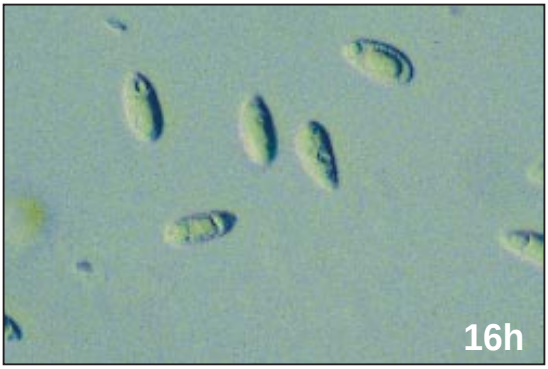


Fig. 16h. Conidia scraped from the surface of a T×D leaf. Photomicrograph (× 1000).

Figs. 16i–m. Leaf blight of black cottonwood and hybrid poplar caused by *Marssonina populi*.

Fig. 16i. Dark angular spots on foliage in a hybrid poplar plantation.

Fig. 16j. Close view of angular spots on hybrid IM214, showing pale orange deposits of conidia (× 25).

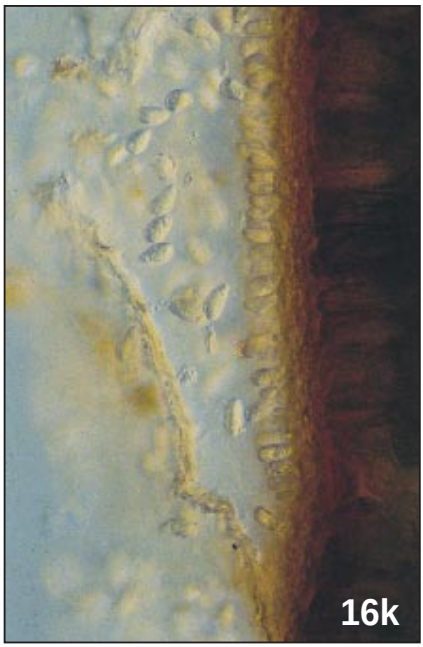


Fig. 16k. Longitudinal section of T×D leaf, showing conidial mass pushing up the epidermis. Photomicrograph (× 500).

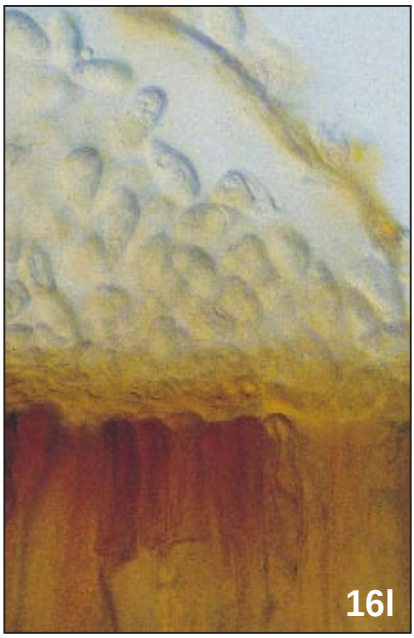


Fig. 16l. Longitudinal section of *P. trichocarpa* leaf, showing conidia under the epidermis. Photomicrograph (× 700).

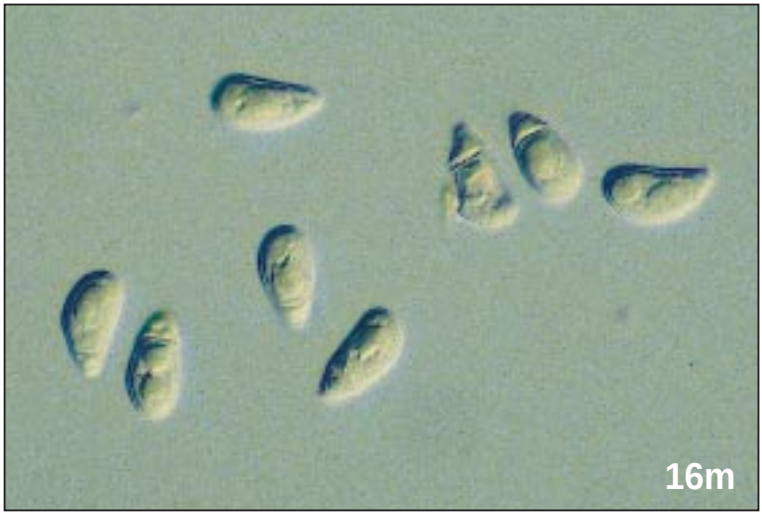


Fig. 16m. Conidia scraped from surface of *P. trichocarpa* leaf. Photomicrograph (× 1000).

Marssonina brunnea f. sp. trepidae: Acervuli are produced on both upper and lower surfaces of the leaves, but are more common on the lower surfaces (Fig. 16a). Conidia are hyaline, with granular contents, egg-shaped or club-shaped, 11–21 × 4–7 µm, one-septate, with an upper, larger rounded cell, and smaller lower cell with a basal scar where it was attached to the conidiogenous cell (Figs. 16b, c). The teleomorph, *D. punctiformis*, has not been properly documented in the province, most likely because it is small and cryptic on dead overwintered leaves.

Marssonina brunnea f. sp. brunnea: Acervuli (Figs. 16f, g) develop on both the upper and lower leaf surfaces. Conidia are narrowly egg-shaped, 15–16 × 5–5.5 µm, straight, hyaline, one-septate with the septum one-third of the spore length from the flattened base (Fig. 16h).

Marssonina castagnei: Acervuli are mostly on the upper surface of the leaf. Conidia are egg-shaped to pear-shaped, 15–23 × 5–8 µm, one-septate, with an upper, larger rounded cell and smaller lower cell with a basal scar where it was attached to the conidiogenous cell.

Marssonina populi: Acervuli mostly occur on the upper surface of the leaf, erupting from the epidermis. Conidia are hyaline, egg-shaped or pear-shaped, 17–27 × 8–13 µm, curved, hyaline, one-septate with smaller cell bearing a flattened scar at the base (Figs. 16k–m). The teleomorph, *D. populorum*, has not been properly documented or recently collected in the province.

Key Diagnostic Features

- angular, often dark lesions on leaf surfaces
- examination of foliar lesions with a hand lens or dissecting microscope reveals dried orange to yellow waxy-looking deposits of conidia
- individual species are separated by conidial morphology and host specificity as previously described

Impact

Marssonina brunnea f. sp. brunnea is the most damaging *Marssonina* on *Populus* worldwide (Spiers 1983; 1988). It has only recently been documented from B.C., where it causes low levels of damage in hybrid poplar trials (Newcombe and Callan 1997). Continued monitoring will be required to determine if this pathogen will become established at significantly damaging levels. The other *Marssonina* species mentioned cause periodic moderate defoliation of aspen and cottonwood throughout the province. *Marssonina populi* causes heaviest damage levels to cottonwoods in the Prince Rupert Region.

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_____. 1988. Comparative studies of type and herbarium specimens of *Marssonina* species pathogenic to poplars. Eur. J. For. Pathol. 18:140–156.

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Leaf Rusts of Cottonwood and Hybrid Poplar

Causal Agents

Melampsora occidentalis H. Jacks. (Basidiomycetes, Uredinales)

M. medusae Thuem. *f. sp. deltoidae*

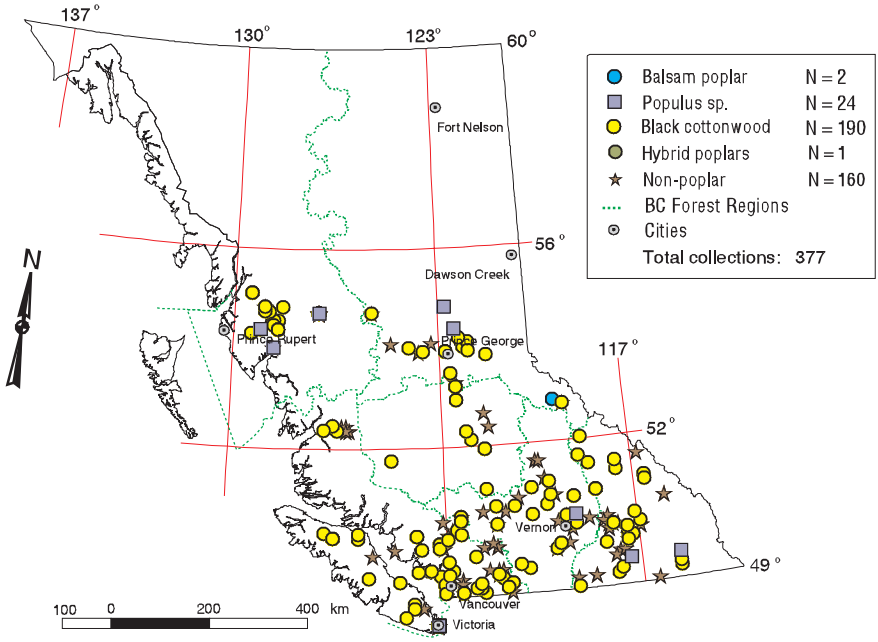
Although the host specificities of these two foliar rusts are different, they produce nearly identical symptoms in the field, and have very similar disease cycles. Therefore, they are discussed together in this section.

Hosts/Host Specificity

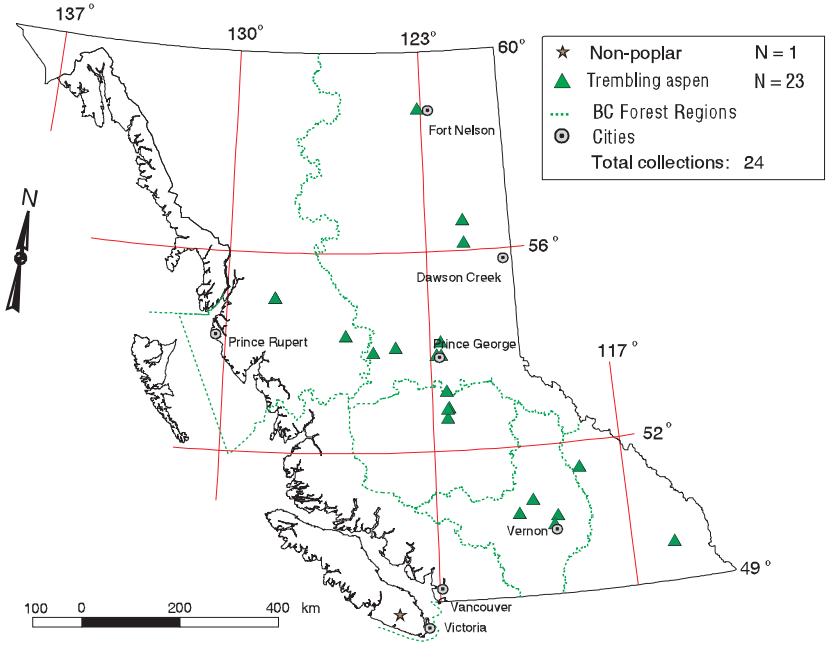
Melampsora occidentalis occurs on *Populus trichocarpa*, *P. balsamifera* (primary hosts), and some hybrid poplars. Although most of the T×D hybrids planted for commercial use in B.C. are resistant to this native rust, occasional flecks or low-level infections occur, especially in plantings surrounded by cottonwoods. *Populus maximowiczii* and its hybrids are also resistant (Hsiang et al. 1993). Alternate (aecial) hosts are in the Pinaceae; in B.C., this includes species of *Abies*, *Larix*, *Picea*, *Pinus*, *Pseudotsuga*, and *Tsuga* (Ziller 1974). Although it is native to western North America, *M. occidentalis* has since spread to *P. trichocarpa* grown in the central United States (Moltzan et al. 1993).

Melampsora medusae f. sp. deltoidae is endemic to eastern North America on *P. deltoides*. On the west coast, it was first discovered in Oregon on hybrid poplars along the lower Columbia River in 1991 (Newcombe and Chastagner 1993a). Two years later, it was detected for the first time in B.C. in commercial hybrid poplar plantations (Fraser Valley) and on susceptible T×D hybrid clones Vancouver Island. The alternate (aecial) host is *Larix* but the aecial stage has not

Distribution of *Melampsora occidentalis.*



Distribution of *Melampsora medusae f. sp. deltoidae.*



yet been collected or confirmed in B.C. Inoculation experiments (Newcombe et al. 1994a) indicate that *Pinus ponderosa* (Douglas) P. Laws. and C. Laws., *P. contorta* (Douglas) Loud. *P. radiata* D. Don, *Pseudotsuga menziesii*, and *L. occidentalis* are aecial hosts.

Time of Appearance

Aecia appear on conifers in spring, and uredinia begin to sporulate on poplar foliage in summer.

Source of Inoculum

Basidiospores from overwintered telia on fallen poplar leaves infect the young conifer foliage in the spring. Aeciospores produced later in the spring on the conifer foliage infect the poplar. Urediniospores produced on poplar foliage during the summer serve to spread and intensify the level of rust infection. Overwintered urediniospores may also reinfect new poplar growth in the spring. The latter phenomenon is suspected to be the primary source of spread in *M. medusae f. sp. deltoidae*. All of these spore stages are wind-borne.

Description

Poplar leaf rusts require two different hosts to complete their life cycles. All endemic species of *Melampsora* rusts on poplars in B.C. (Fig. 17a) have an alternate conifer host. Basidiospores from overwintered telia on fallen poplar leaves infect young conifer foliage in the spring, and small orange to yellow aecial pustules appear approximately two weeks later (Fig. 17i). Symptoms of needle necrosis and discoloration due to *M. occidentalis* are most striking in *Pseudotsuga* (Fig. 17j) and *Larix*. Aeciospores of *M. occidentalis* are hyaline, subglobose, 22–27 × 26–35 µm, finely warty, and thickened bilaterally on opposite sides. These spores infect poplar leaves in the summer, and 2 weeks after infection orange uredinial pustules begin to appear on the foliage (Figs. 17b, c). Urediniospores are ellipsoid to pyriform, 16–29 × 32–48 µm, with evenly distributed spines over the spore surface and bilaterally thickened spore walls (Figs. 17d, e).

In late summer and early fall, uredinial production falls off and in their place, brown, cushion-like telia are produced (Fig. 17f), consisting of tightly packed, apically thickened teliospores, 40–64 × 10–20 µm (Fig. 17g).

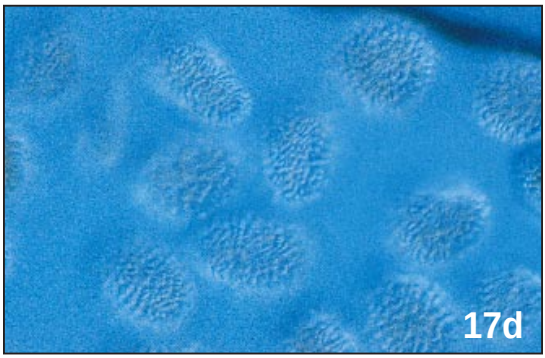
Mempsora medusae f. sp. deltoidae is indistinguishable from *M. occidentalis* in the field (Figs. 17j–l), apart from having different host specificities. Separation of the two taxa in B.C. is based on differences in urediniospore and teliospore morphology. Urediniospores of *M. medusae f. sp. deltoidae*, measuring from 26–37 × 15–22 µm, are considerably smaller on average than *M. occidentalis*, (Fig. 17n) and with the additional distinction of a smooth equatorial patch free of spines (Fig. 17o). However, the two species appear to be hybridizing; thus, it will become much harder to tell them apart (G. Newcombe, Washington State University, pers. comm.). Apart from inoculation studies, *Melampsora medusae f. sp. deltoidae* has not been observed infecting conifers in the Pacific Northwest (Newcombe et al. 1996).

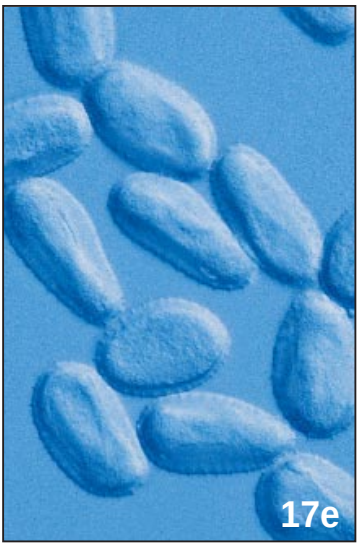
Figs. 17a–i. *Melampsora occidentalis* on cottonwoods and their alternate hosts.

Fig. 17a. Rusted vs. resistant T×D hybrid clones in a Vancouver Island plantation.

Fig. 17b. Uredinial sporulation on cottonwood.

Fig. 17c. Uredinia on the undersurface of a cottonwood leaf.





Figs. 17d–e. Urediniospores, showing spines in optical section (Fig. d) and surface ornamentation (Fig. e). Photomicrographs (× 600; × 700).

Fig. 17f. Telia on cottonwood.

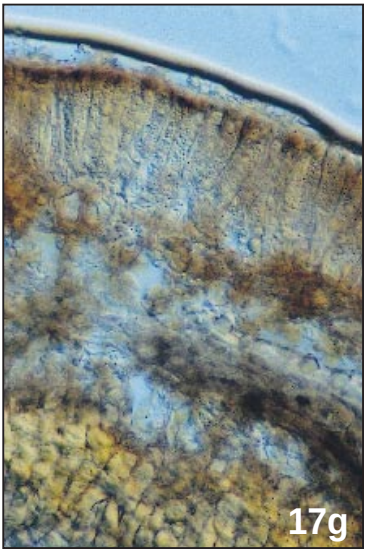


Fig. 17g. Teliospores from a sectioned cottonwood leaf. Photomicrograph (× 200).

Fig. 17h. Needle discoloration of Douglas-fir infected with *M. occidentalis*.

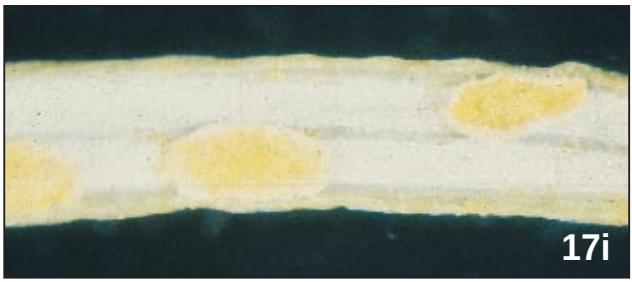


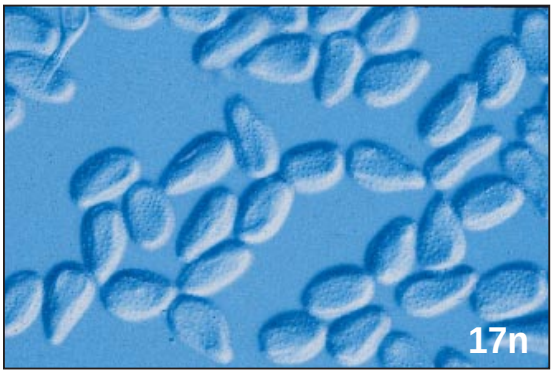
Fig. 17i. Aecia on Douglas-fir needles.

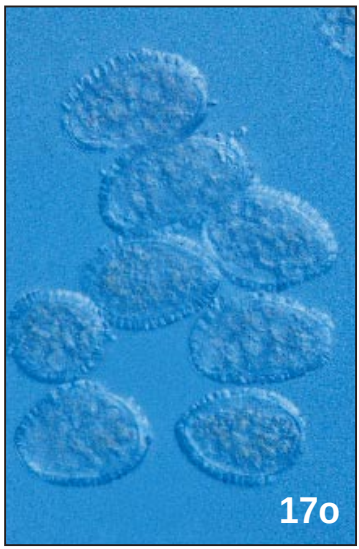
Figs. 17j–o. *Melampsora medusae f.sp. deltoidae* on hybrid poplars.

Fig. 17j. Susceptible, rusted T×D hybrid poplar planted in the foreground; resistant clone of the same age in background.

Figs. 17k, l. Heavy uredinial sporulation on susceptible T×D.

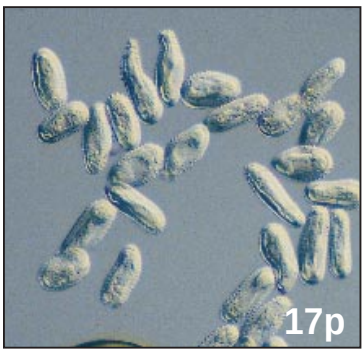
Fig. 17m. Uredinial pustules on susceptible T×D hybrid.





Figs. 17n, o. Urediniospores scraped from pustules on T×D hybrid, showing surface ornamentation (Fig. n) and bald spots (Fig. o). Photomicrographs (× 400; × 800).

Figs. 17p-q. *Melampsora larici-populina*.



Figs. 17p, q. Urediniospores of *Melampsora larici-populina* from herbarium specimen of *Populus trichocarpa* var. *hastata*. Photomicrographs (× 150; × 800).

Key Diagnostic Feature

- masses of orange powdery uredinial pustules on infected poplar leaves

Look-alikes

Although it has not yet been detected in B.C., *M. larici-populina* Kleb. has been introduced to hybrid poplar plantations in Oregon, Washington, and California (Newcombe and Chastagner 1993b). It is separable from the two species previously described based on urediniospores which are elongated, 30–50 × 13–22 µm, with a smooth, spine-free apex (Figs. 17p, q). Newcombe et al. (1994) showed via artificial inoculation that *Pinus ponderosa* and *P. contorta* are aecial hosts. *Larix decidua* Mill., *L. occidentalis*, *L. leptolepis* (Siebold & Zucc.) Gord., and *Pinus radiata* D. Don. are also hosts (Shain 1988). This rust is also capable of hybridizing with *M. medusae* (Spiers and Hopcroft 1994).

Melampsora populnea (Pers.) P. Karst. occurs on *P. alba* in B.C., where it was introduced from Europe. It is distinct in being exclusive to this host, by not producing a telial stage, and by urediniospores measuring 20–30 × 15–22 µm, without bilateral thickenings.

Impact

Leaf rusts caused by *Melampsora* species cause premature defoliation and are very damaging and important pathogens of poplars. Repeated severe early leaf loss in cottonwood due to *M. occidentalis* can reduce growth and yield during the following growing season. Such losses in southern B.C. have been documented by Wang and Van Der Kamp (1992) and in Washington by Dunlap et al. (1994) and Newcombe et al. (1994b). In the north-central United States, leaf rusts have been shown to reduce wood volume an average of 29–32% and dry weight an average of 31–42%. To date the only practical way to control poplar leaf rust has been through host resistance. Resistance in *P. trichocarpa* to *M. occidentalis* is known to be race-nonspecific (Hsiang and Chastagner 1993; Hsiang et al. 1993). Major genes for resistance to *M. medusae f. sp. deltoidae* have already been elucidated (Newcombe et al. 1996). Many hybrid poplar clones are susceptible at some level to *M. occidentalis* and *M. medusae f. sp. deltoidae*. Mixed uredinial infections on the same leaf provide ideal conditions for hybridization, and this phenomenon has already been documented between other rust species (Spiers and Hopcroft 1994). Field and microscopic evidence indicates that this is also happening between *M. occidentalis* and *M. medusae f. sp. deltoidae* (George Newcombe, pers. comm.).

Regular microscopic examination of rusts will aid in the detection of spore morphology intermediate (hybrid) between the two rust species (Spiers and Hopcroft 1994). Early detection of changes in rust populations may aid in prediction of future yield losses. Planting of clonal mixtures will also help to prevent rusts from overcoming host resistance (Spiers and Hopcroft 1994).

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Leaf Rust of Aspen

Causal Agent

Melampsora medusae Theum. *f. sp. tremuloidae* (*sensu* Ziller, 1955, 1965; = *M. albertensis* Arth., *sensu* Allen et al. 1996 and Hiratsuka et al. 1995) (Basidiomycetes, Uredinales)

There is some question as to whether *M. medusae* and *M. albertensis* are the same species. Additional inoculation studies on the aecial hosts are required to fully understand this species or species complex. Hiratsuka et al. (1995) believe that true *M. medusae* is an eastern North American species, with a natural range that only extends as far west as eastern Manitoba.

Hosts/Host Specificity

This rust is limited to *P. tremuloides* in B.C., and alternate hosts, which are *Abies*, *Larix*, *Picea*, *Pinus*, *Pseudotsuga*, and *Tsuga*. *Larix* and *Pseudotsuga* are the most common aecial hosts. Shain (1988) demonstrated that urediniospores from aspen will not infect eastern cottonwood (*P. deltoides*), and based on these findings, proposed the separation of *M. medusae* into two *formae speciales*.

Time of Appearance

See preceding description for leaf rusts of cottonwood.

Source of Inoculum

See preceding description for leaf rusts of cottonwood.

Distribution of *Melampsora medusae f. sp. tremuloidae.*

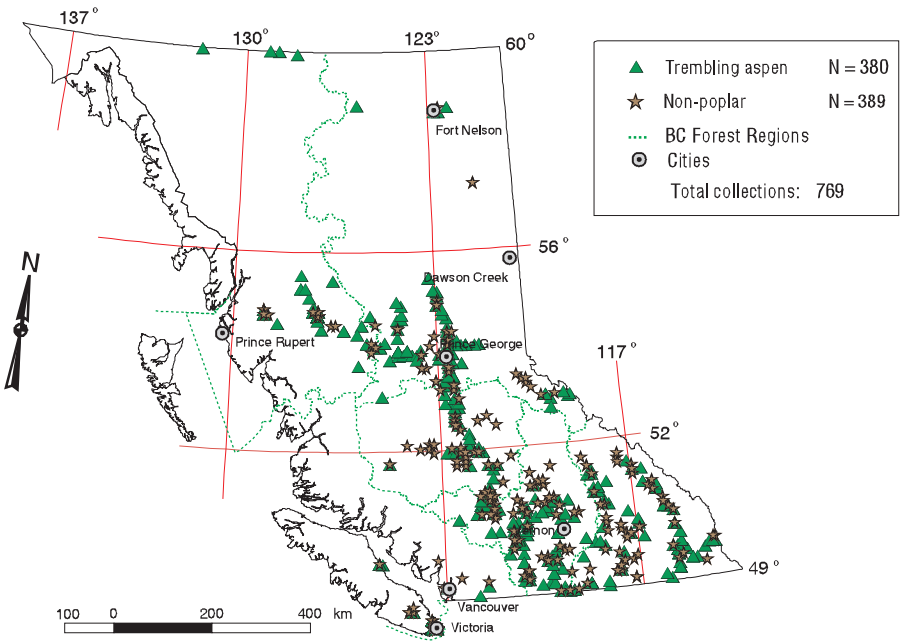

Fig. 18. *Melampsora medusae* on trembling aspen.

Fig. 18a. Heavy uredinial sporulation on trembling aspen.

Fig. 18b. Uredinia (powdery areas) and developing telia (smooth orange areas) on the abaxial leaf surface.

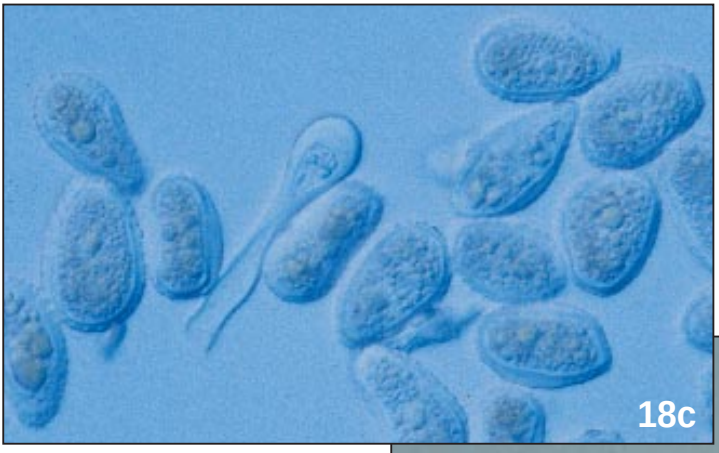

Fig. 18c. Urediniospores and capitate paraphyses. Photomicrograph (× 600).

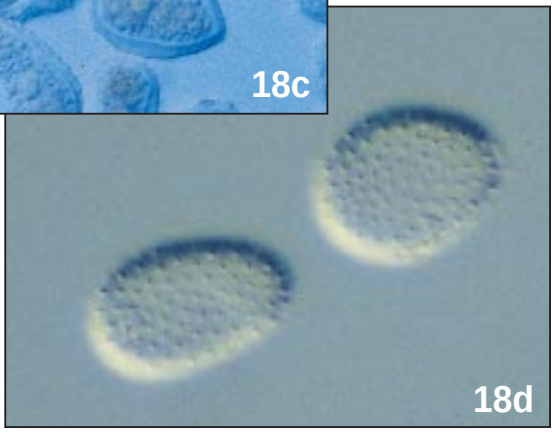

Fig. 18d. Surface ornamentation (spines) on urediniospores. Photomicrograph (× 900).

Description

See description of leaf rusts of cottonwood for general field symptoms. Host alternation appears to be obligatory for this species. *Melampsora medusae f. sp. tremuloidae* differs from other rust taxa in uredinial (Figs. 18a, b) host specificity, and in smaller urediniospore size, which is 23–35 × 15–23 µm (Figs. 18 c, d). Teliospores are cinnamon-brown, 29–45 × 10–15 µm (Ziller 1974).

Key Diagnostic Feature

- orange powdery uredinia on aspen leaves

Impact

This rust causes more damage to seedlings of the coniferous hosts, particularly *Pseudotsuga* and *Larix*. It is associated with low to moderate levels of aspen defoliation throughout its range in B.C.

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Septoria Leaf Blight of Cottonwoods and Hybrid Poplars

Causal Agent

Septoria populicola Peck (teleomorph = *Mycosphaerella populicola* G.E. Thompson) (Ascomycetes, Dothideales)

Hosts/Host Specificity

Populus trichocarpa is the most commonly affected native host in B.C., while *P. balsamifera*, T×D, and T×M hybrids are also often highly susceptible. *Populus tremuloides* is rarely infected.

Time of Appearance

Leaf spots first develop in late spring or early summer, and are especially common in years with wet mild springs.

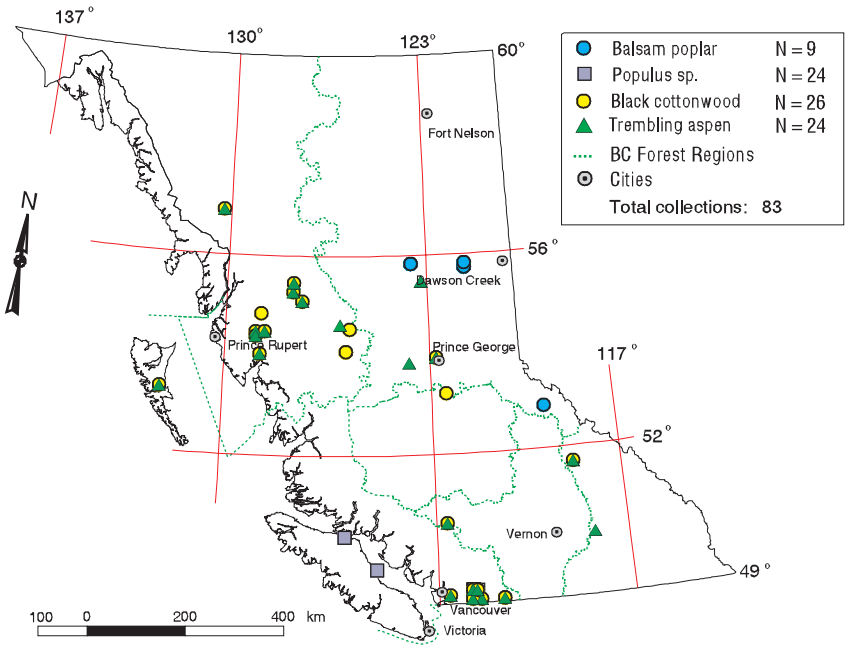
Source of Inoculum

Ascospores ejected from overwintered leaves bearing pseudothecia may play a role in initiation of spring infections in B.C.; their role in the disease cycle of septoria canker is well-known (Luley and McNabb 1989). Overwintered conidia also provide primary inoculum. Secondary spread during the growing season is from water-splashed conidia, which ooze out of pycnidia in tendrils during periods of high humidity.

Description

Septoria populicola is only found in association with leaf spots in B.C., but in eastern Canada it may also be associated with stem cankers on *P. deltoides* and susceptible hybrids (Thompson 1941; Zalasky 1978).

Distribution of *Septoria populicola.*



The presence of the sexual state has not been well documented in B.C., and it is not known how frequently it is produced. Preliminary examination of fallen leaves under infected trees have shown that most *Mycosphaerella* fruiting bodies present are those of a common saprophyte, *M. tassiana* (De Not.) Johans. (B. Callan, Canadian Forest Service, Victoria, B.C., unpublished data). Pseudothecia of *M. populicola* are globose, 96–160 µm in diameter, produced in masses in overwintered dead fallen leaves. Asci are hyaline, cylindrical to clavate, 64–110 × 13–16 µm. Ascospores are hyaline, fusiform, one septate, 22–32 × 6–6.5 µm.

Infections on young leaves are first evident in early summer when yellow lesions begin to develop on leaves (Figs. 19a, d), soon turning dark brown to black (Figs. 19b, c). Host response depends upon the clone; some very susceptible trees show pronounced foliar yellowing. Leaf spots have distinct, rounded margins, which are never feathery or angular; the spots often form in lines parallel to the long axis of the leaf. This pattern is presumably due to the spread of conidia along the trail of a water droplet as it rolls down to the leaf tip. Globose, black, ostiolate pycnidia, 48–128 µm in diameter (Figs. 19e, f), develop within the discolored areas of the leaves; they are most common on the upper surface, but may also be found on lower surfaces. Pycnidia ooze pinkish tendrils of conidia during periods of high humidity (Fig. 19g). Conidia are hyaline, long, sinuous, 60–110 × 3.5–4.5 µm, 3–6 septate (Fig. 19h).

Key Diagnostic Features

- rounded lesions on leaves
- yellow discoloration of foliage, premature defoliation
- extremely long and narrow conidia ooze from pycnidia in pink tendrils during wet weather

Look-alikes

Mycosphaerella tassiana (De Not.) Johan. is a common saprophytic inhabitant of fallen, overwintered cottonwood leaves; its pseudothecia may be easily confused with those of *M. populicola* until they are examined microscopically. The ascospores of *M. tassiana* are larger than the dimensions reported for *M. populicola*, measuring 16–29 × 4.5–8 µm (Corlett 1991). Additionally, the anamorphs of the two species are strikingly different. The conidial state of *M. tassiana*, which is formed within 3 days after ascospore germination in culture (Barr 1958), is *Cladosporium herbarum* (Pers.:Fr.) Link, a cosmopolitan olive-green mold (see the section on *Venturia populina* for illustration of the asexual stage).

Septoria musiva Peck (teleomorph = *Mycosphaerella populorum* G.E. Thompson) does not occur in the Pacific Northwest, according to field surveys and examination of regional herbarium specimens (Newcombe et al. 1995). *Septoria musiva* is associated with severe canker damage in the midwest and on the east coast. Pseudothecia are globose, 60–110 µm in diameter, black, ostiolate, and immersed in fallen overwintered leaves and cankered stems. Asci are hyaline, clavate, 54–70 × 13–16 µm. Ascospores are hyaline, fusiform, uniseptate, 16–28 × 4.5–6 µm, slightly narrower than those of *M. tassiana*, but the two species are readily separable by their strikingly different anamorphs. *Septoria musiva* pycnidia are immersed in leaves, and cankered tissue, globose, 48–128 µm in diameter, and ooze pinkish tendrils of conidia during wet weather. Conidia are hyaline, cylindrical, one to four septate, 28–54 × 4 µm – much smaller than those of *S. populicola*.

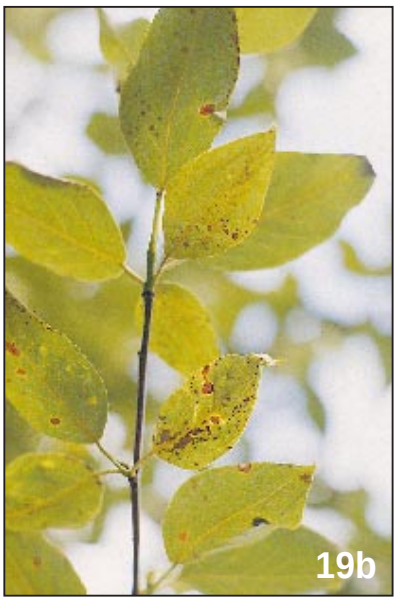
Impact

Epidemics of severe foliar discoloration and premature defoliation due to leaf spot in the lower Fraser Valley and on Vancouver Island have been documented in recent years (Newcombe et al. 1995). After Melampsora rusts, Septoria leaf spot is probably the next most damaging foliar disease of black cottonwood and susceptible hybrid poplars in B.C.

Control methods of poplar leaf spots in nurseries were outlined by Carlson (1974); resistance in eastern cottonwood has also been documented (Cooper and Filer 1976).

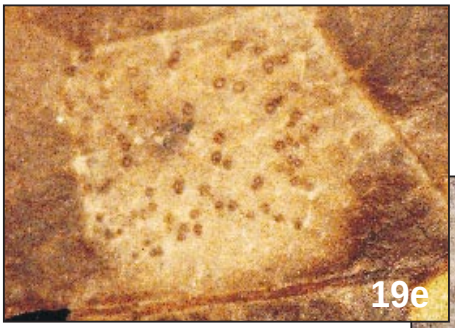
Fig. 19. *Septoria* leaf blight of cottonwood and hybrid poplars.

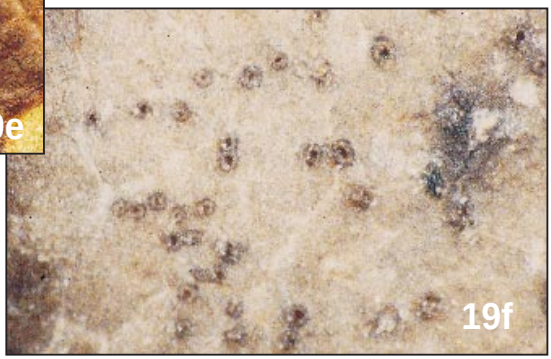
Fig. 19a. Leaf spots on T×M hybrid poplar.



Figs. 19b, c. Symptoms on T×D hybrid poplars (dark spots, fused in some areas along leaf margins).

Fig. 19d. Chlorosis of susceptible T×D foliage precedes darker necrotic spots. At this point in the development of the disease, sporulating fruiting bodies are present in the central necrotic flecks.





Figs. 19e, f. Pycnidia of *Septoria populicola* on black cottonwood (× 15; × 35).

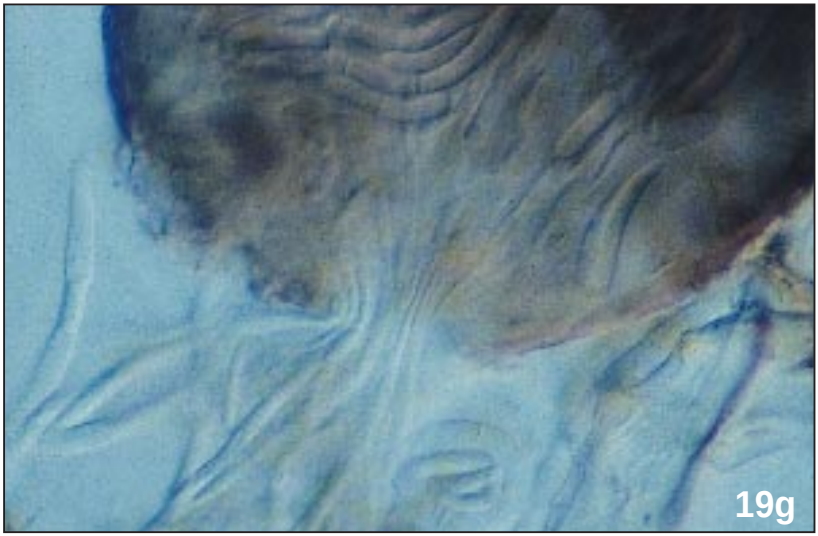


Fig. 19g. Conidia extruding from pycnidium. Photomicrograph (× 850).

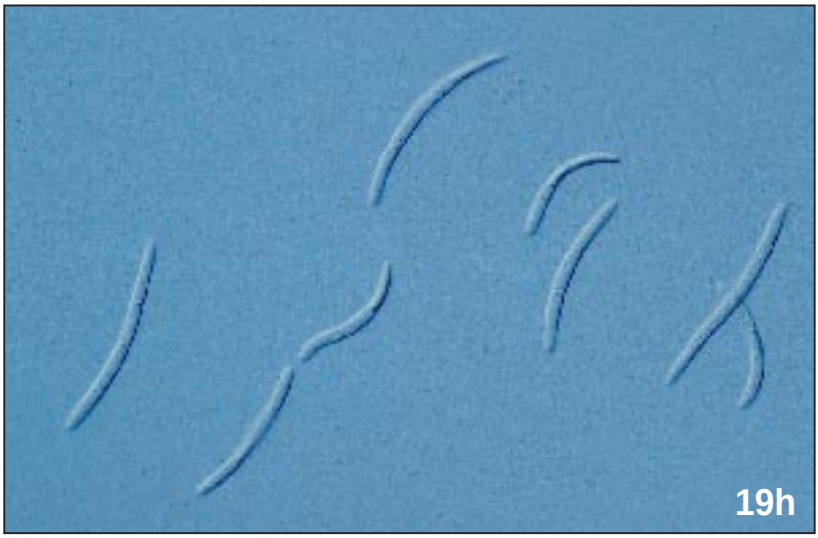


Fig. 19h. Conidia. Photomicrograph (× 600).

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Taphrina Leaf Blister and Leaf Curl of Poplars

Causal Agents

Taphrina populina (Fr.:Fr.) Fr. and *T. populi-salicis* Mix (Ascomycetes, Taphrinales)

Hosts/Host Specificity

Taphrina populina occurs on *P. trichocarpa*, T×D, T×N hybrids, and *P. tremuloides* in B.C.;*Taphrina populi-salicis* is limited to *P. trichocarpa* and *Salix* spp.

Time of Appearance

Blistered areas appear in the spring on young leaves.

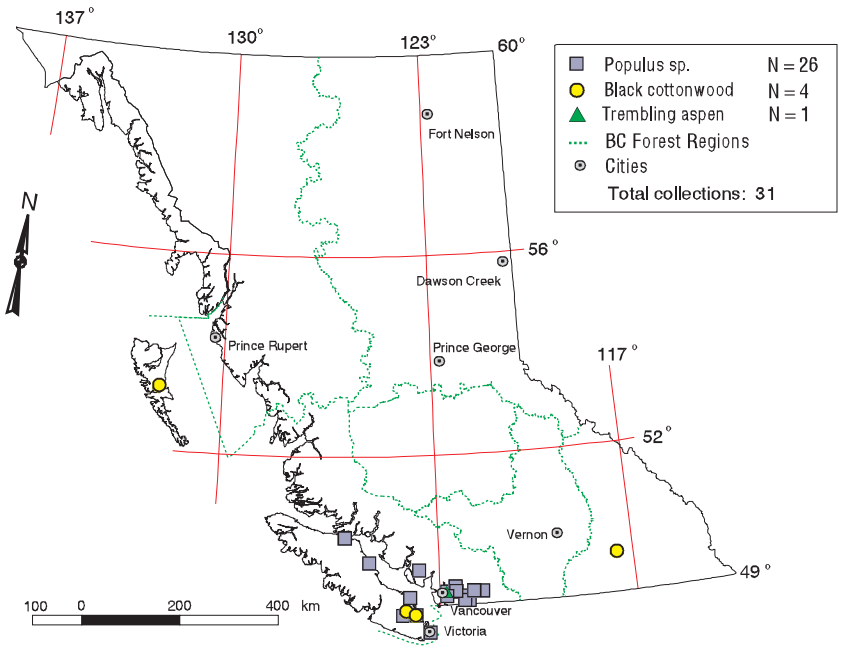
Source of Inoculum

Taphrina may live saprophytically in a yeast-like phase, overwintering on dormant bark and buds. Spores from the yeast phase are spread by wind and rain during the spring after bud break, when they infect young leaves before the cuticle has hardened (Mix 1949).

Description

The first signs of infection are swollen, cupped, round to oval blisters, 0.5 to several cm in diameter on leaves in the spring (Fig. 20a). Blisters form with the cupped side facing down (only rarely does the reverse occur on the upper surface of the leaf). The blisters turn golden yellow as asci mature on the leaf undersurface (Fig. 20b); infected areas may be colonized by secondary molds and yeasts, becoming reddish or blackened late in the growing season. In dense, high humidity plantings such as stool beds, entire leaves may be overcome and turned into a single yellow "blister." Asci are naked (no fruiting bodies) and form a single tightly packed layer on the yellow blister surface.

Distribution of *Taphrina populina*.



Distribution of *Taphrina populi-salicis.*

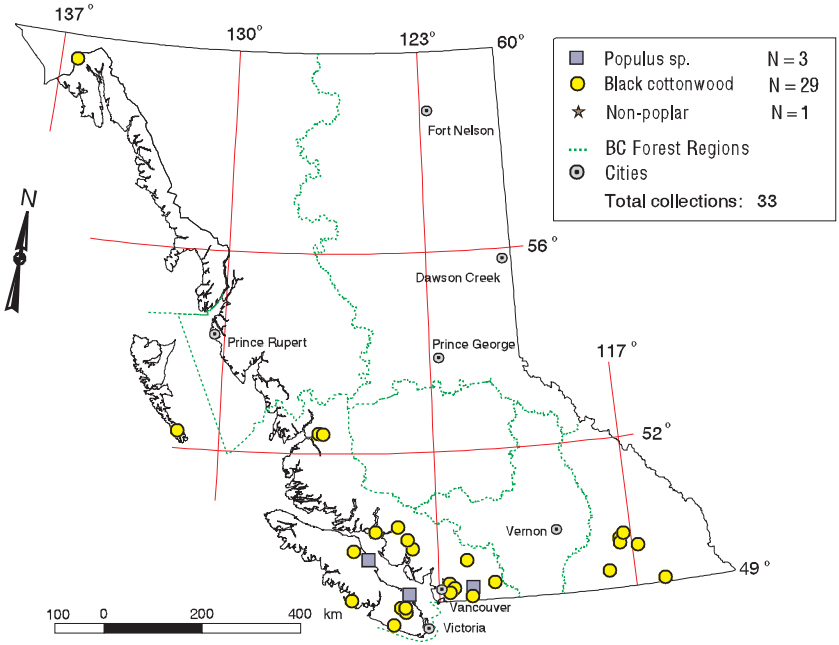


Fig. 20. *Taphrina* leaf spots on cottonwoods and hybrid poplars.

Fig. 20a. *Taphrina populina* sporulating in large yellow deformed areas on T×D hybrid poplar leaves.

Fig. 20b. Yellow spots associated with *T. populina* infections on a T×M hybrid poplar.

Fig. 20c. Yellow-pigmented asci of *T. populi-salicis* on cottonwood. Photomicrograph (× 600).

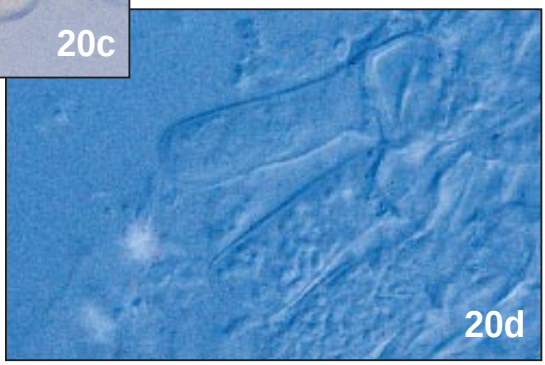


Fig. 20d. Asci, foot cell and masses of small, ellipsoid ascospores of *T. populina* on T×D hybrid poplar. Photomicrograph (× 700).

The two species of *Taphrina* are difficult to distinguish, but may be separated based on ascus and ascospore size range, and to a lesser extent, host preference. Asci, which occur in a densely packed layer on blisters, are cylindrical, containing hundreds of spores which form by budding. Prior to ascospore development, asci contain a bright yellow, oily-looking cytoplasm (Fig. 20c). Asci of *T. populina* measure 30–122 × 13–30 µm, and ascospores, which are hyaline, round, and single-celled, range from 4–6.5 × 4–5 µm (Fig. 20d). Asci of *T. populi-salicis* measure 50–106 × 13–30 µm, and ascospores, which are hyaline and round, are very small, 1.5–5 × 0.5–4.5 µm (Funk 1985).

Key Diagnostic Features

- golden yellow leaf blisters
- single palisade of many-spored asci on surface of blisters

Look-alikes

Certain aphid and mite infestations also cause brightly colored twisted foliage, but signs of their presence (frass, cast skin, webs) are usually present.

Impact

Both species occur at low levels in natural stands and in hybrid poplar plantations in B.C.

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Aspen Shoot and Leaf Blight

Causal Agent

Venturia macularis (Fr.:Fr.) E. Müller & Arx (Ascomycetes, Venturiaceae) Anamorph: *Pollaccia radiosa* (Lib.) Baldacci & Cif. according to Allen et al. (1996); *P. americana* Ondrej according to Hunt (1978). Much of the current taxonomic literature on *Venturia* is based upon European type material and hosts (Morelet 1985), and may not represent the situation in western Canada. A comprehensive examination of *Venturia* on poplar in western North America would likely result in the separation of the *V. macularis* complex into several closely related but distinct taxa, some of which could be unique to this continent. The relatively recent discovery of *Pollacia borealis* Funk on aspen in North America (Funk 1989a, b) demonstrates the need for future study of this species complex.

Hosts/Host Specificity

Populus tremuloides is the primary host in B.C. Elsewhere in North America *V. macularis* is also associated with *P. grandidentata*, *P. alba*, *P. deltoides*, and various of their hybrids (Barr 1968).

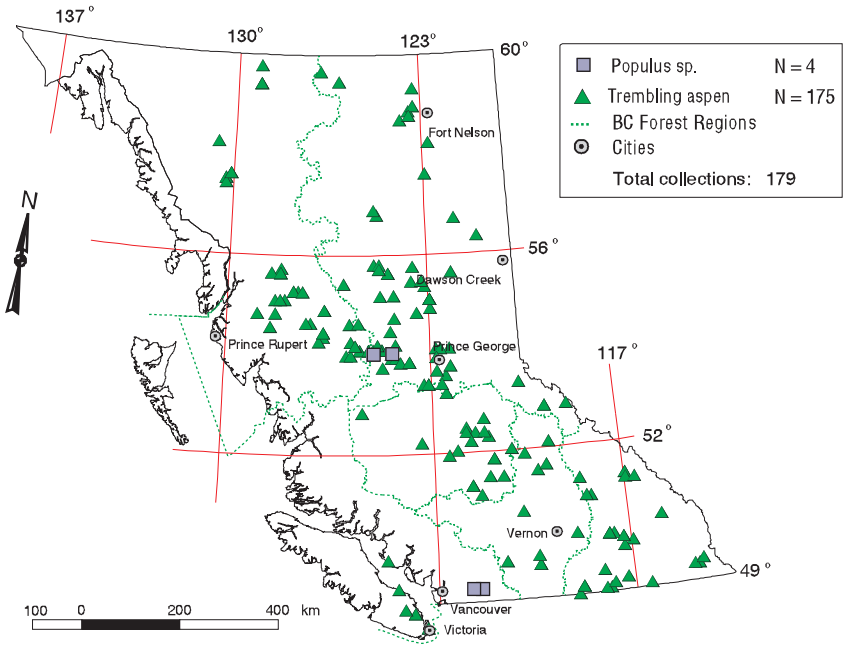
Time of Appearance

New infections begin in the spring, during bud break and leaf expansion.

Source of Inoculum

Each spring, primary inoculum is initiated from two sources: wind-borne ascospores forcibly ejected from overwintered pseudothecia on dead shoots and leaves, and overwintered conidia on blighted twigs. Secondary inoculum is from rain-splashed conidia produced on dead shoots and leaf spots in spring and summer.

Distribution of *Venturia macularis*.



Description

New growth in the spring is infected by airborne ascospores or rain-splashed conidia. Periods of mild wet spring weather are conducive to epidemics, and the first black spots of the season are often adjacent to the shriveled grey remnants of last year's shepherd's crooks (Fig. 21d, e). Pseudothecia (overwintering ascomata) are produced late in the growing season on fallen dead leaves and old shepherd's crooks, the latter often remaining attached to the tree until the following spring. Pseudothecia are globose, smooth to setose, 0.08–0.14 mm in diameter, erumpent through the host epidermis, with an apical ostiole 0.02–0.05 mm in diameter. Setae when present average 30–50 µm in length. Asci are cylindrical to oblong, 4–63 × 10–12 µm, bitunicate, bearing two, four, or eight olive-brown ascospores (Fig. 21i). These ascospores are faintly rough-walled to smooth, elliptical to clavate, 8–14 × 4–6 µm, unequally two-celled.

After infection by ascospores or overwintered conidia, diseased tissues wilt and turn black, and the dead areas become covered with an olive green velvety coating of conidiophores and conidia by early summer (Fig. 21f). Conidiogenous cells are cylindrical, olive green to brown, non-septate, and 8–12 × 4–6 µm (Fig. 21g). Conidia (Fig. 21h) are olive-brown, ellipsoid to cylindrical, 12–22 × 6–7 µm, straight or slightly curved, 0–2 septate (Funk 1985).

Key Diagnostic Features

- terminal shoots and new growth on branch tips of aspen become blackened and shriveled in spring and early summer
- blackened, twisted lesions on young aspen leaves
- velvety mats of olive green conidia on leaf spots and shepherd's crooks
- stunted, malformed understorey trees with thin foliage

Look-alikes

Pollaccia borealis Funk is associated with a purple-brown leaf spot of aspen (Fig. 21j) in northern B.C. and the Yukon Territory (Funk 1989a). The small circular spots average less than 0.5 cm in diameter, and have a slightly raised darker margin, which is clearly visible on the undersurface of the leaf (Fig. 21k). The conidia are borne only on the lower surface of the lesion, while a network of dark, branching mycelium is visible through the epidermis on the upper side. The infected areas frequently detach and fall out, causing a "shot-hole" symptom on the infected leaf. Conidiogenous cells are cylindrical, hyaline, and produce light brown, cylindrical aseptate conidia averaging 15–22 × 4–5 µm.

Pollaccia borealis may occur on leaves also infected with *V. macularis*, but the two species sporulate in separate lesions. The former species is distinguished by the paler purplish lesions; narrower, nonseptate conidia; and the fact that it is not associated with the more severe symptom of shoot dieback (Funk 1989a). The sexual state of *P. borealis*, *Venturia borealis* Funk (Fig. 21l) has not yet been observed in nature, but has been produced in and described from culture (Funk 1989b).

Impact

In years with moist mild spring weather most shoots in young aspen stands naturally regenerated by sprouting may die (Fig. 21a). Repeated infections by *V. macularis* may result in small, stunted, deformed crowns (Figs. 21b, c). This damage is most commonly reported from the Prince George and Prince Rupert Districts (Hunt 1978).

Figs. 21a–i. Shoot and leaf blight of trembling aspen, caused by *V. macularis*.

Fig. 21a. Aerial view of severely blighted aspen stand.

Fig. 21b. Thin crowns of infected aspen.

Fig. 21c. Discolored blighted foliage.

Figs. 21d, e. Characteristic shepherd's crooks associated with shoot tip dieback.

Fig. 21f. Olive green masses of conidia on the surface of a dead aspen leaf.

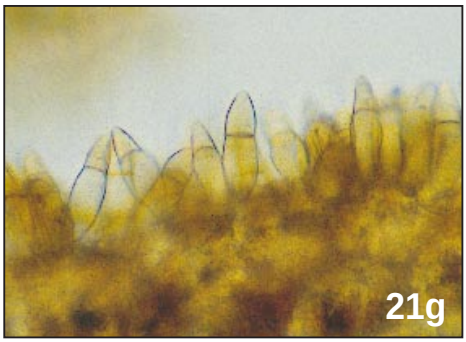


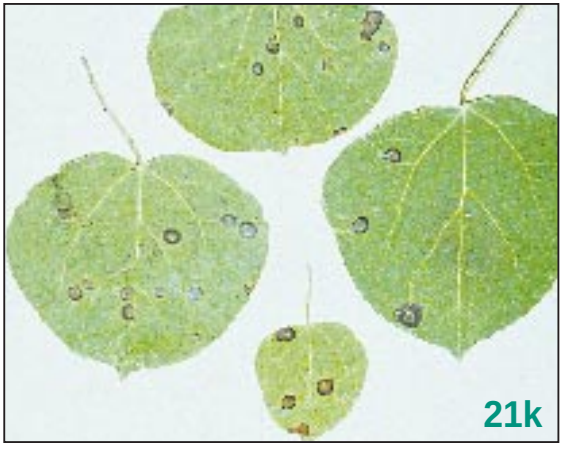
Fig. 21g. Conidial mass of *Venturia macularis* on an aspen leaf section. Photomicrograph (× 600).

Fig. 21h. Conidia. Photomicrograph (× 1200).

Fig. 21i. Asci and ascospores from overwintered pseudothecium on previous years' shepherd's crook. Photomicrograph (× 1200).

Figs. 21j–l. Leaf spot of aspen caused by *Pollaccia borealis.*

Fig. 21 j–k. Small purplish foliar lesions on aspen.



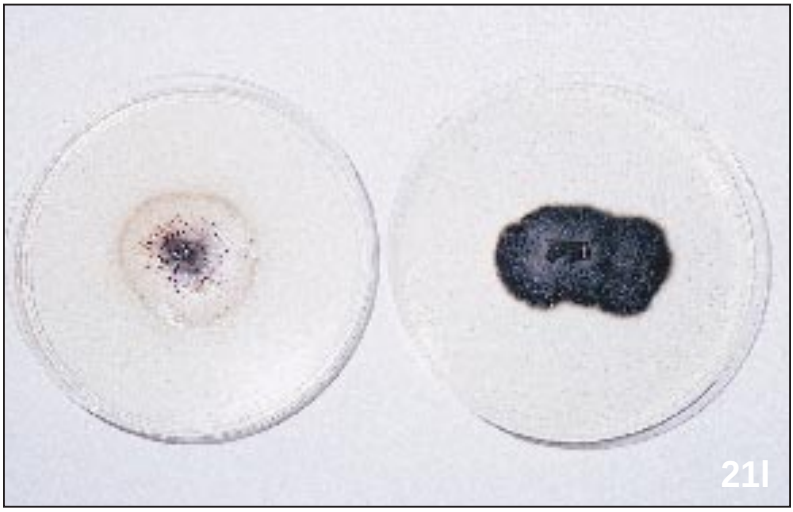


Fig. 21l. The teleomorph *Venturia borealis* is known only from culture. Pseudothecia (left), are growing on cornmeal agar; conidia are produced on malt agar (right).

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Poplar Leaf and Shoot Blight

Causal Agent

Venturia populina (Vuill.) L. Fabricius (Ascomycetes, Venturiaceae) (anamorph = *Pollaccia elegans* Servazzi)

Hosts/Host Specificity

In B.C., *P. trichocarpa*, *P. balsamifera* and many of the T×D hybrids are susceptible; the other hybrids in section *Tacamahaca* are less susceptible. Some commercially utilized T×D clones such as 49–177 and T×M hybrids are known to be resistant; in the latter case the resistance is conferred by the *P. maximowiczii* parent. Elsewhere in North America, disease incidence records are from poplars in section *Tacamahaca*, while in Europe and India, poplars in *Aigeiros* are also reported as hosts. Records in the Pacific Northwest of *V. populina* begin in 1973 (Newcombe and van Oosten 1997).

Time of Appearance

Dead shoots (Figs. 22a–c) and black leaf spots become apparent in susceptible trees by mid-June in damp coastal regions. Secondary outbreaks may also occur in summer and fall following wet periods.

Source of Inoculum

Each spring, primary inoculum is initiated from two sources: wind-borne ascospores forcibly ejected from overwintered pseudothecia (Figs. 22g, h) on dead shoots and leaves, and overwintered conidia on blighted twigs. Secondary inoculum is from rain-splashed conidia produced on dead shoots and leaf spots in spring and summer.

Distribution of *Venturia populina*.

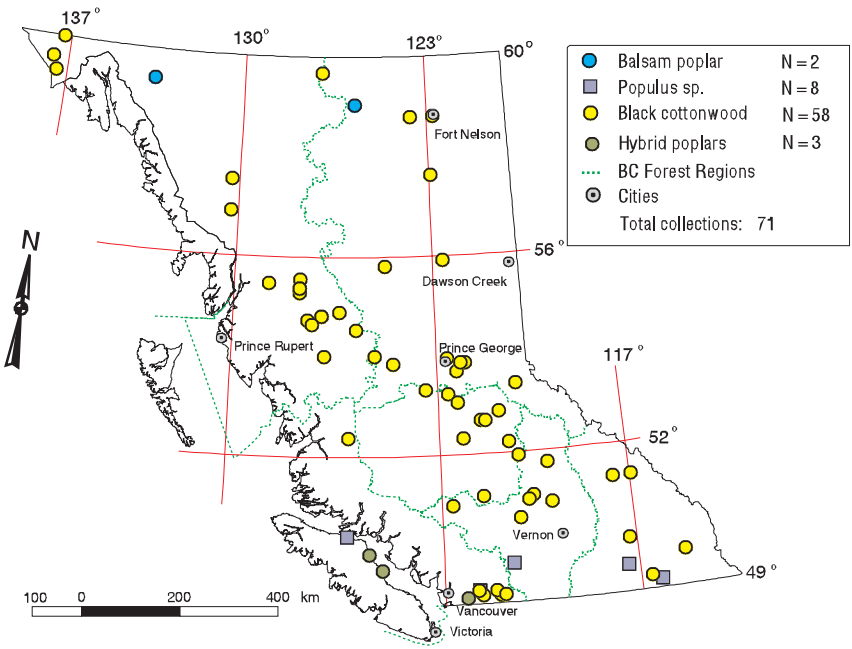

Fig. 22. *Venturia* blight of cottonwood and hybrid poplars.

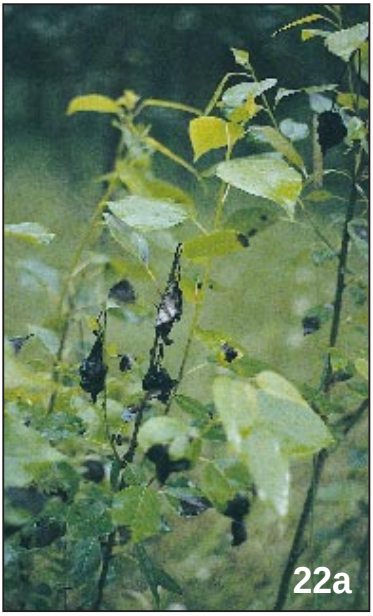

Fig. 22a. Shepherd's crooks on T×D hybrid poplars.

Fig. 22b. Dieback from leaf petiole, developing into a canker on the succulent main stem of a T×D sapling.

Fig. 22c. Stunted, deformed susceptible T×D clones on Vancouver Island.

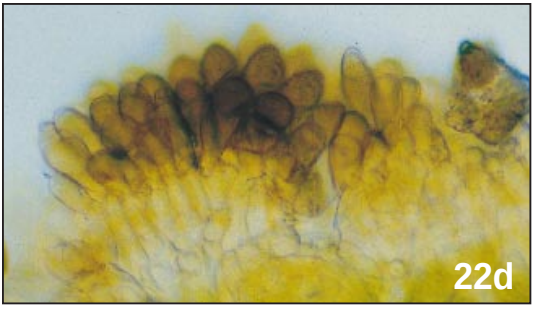

Fig. 22d. Conidial palisade of *V. populina* on a living cottonwood leaf. Photomicrograph (× 600).

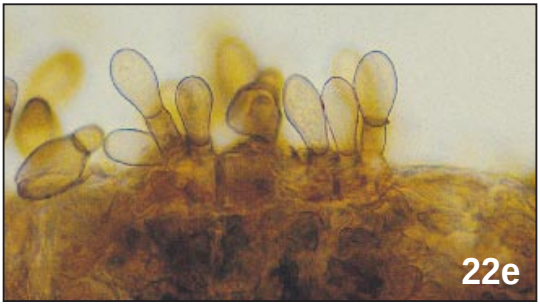

Fig. 22e. Developing *V. populina* conidia on a living T×D hybrid poplar leaf. Photomicrograph (× 800).

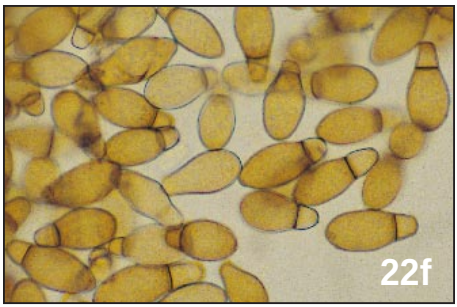

Fig. 22f. Mature conidia scraped from a T×D hybrid poplar leaf. Photomicrograph (× 600).

Fig. 22g. Overwintered pseudothecia (black spots) protruding from a dead cottonwood leaf and petiole.

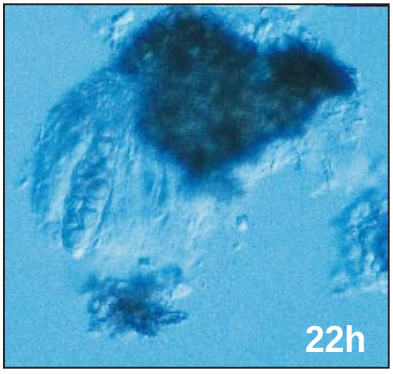

Fig. 22h. Portion of crushed pseudothecium, showing asci. Photomicrograph (× 250).

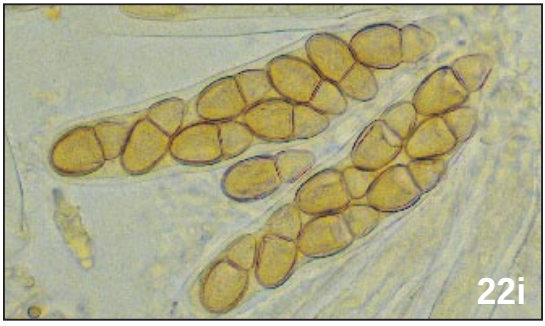

Fig. 22i. Asci with mature ascospores. Photomicrograph (× 600).

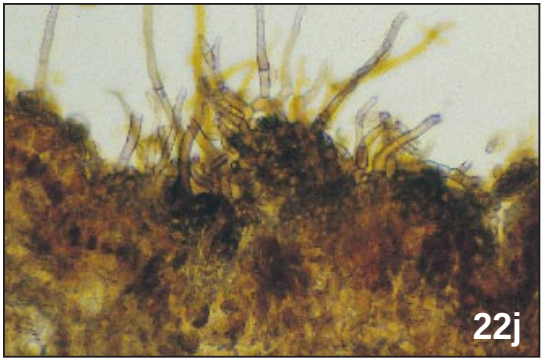

Fig. 22j. Conidiophores of *Cladosporium* sp., which are longer, more branched and more irregular than those of *Venturia* spp., on poplar. Photomicrograph (× 100).

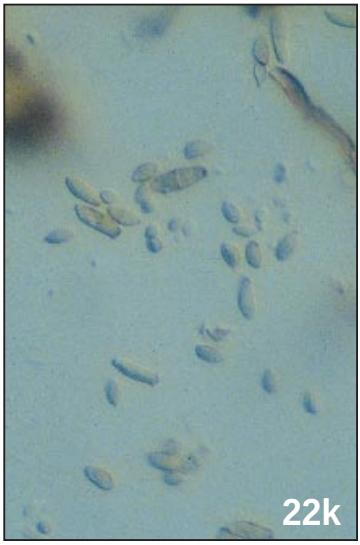

Fig. 22k. Conidia of *Cladosporium*. Note irregular septation, size variation, and multiple pores present on some conidia. Photomicrograph (× 150).

Description

The disease cycle and description of shoot dieback and foliar spots is identical to that described for *V. macularis* (Hunt 1978). *Venturia populina* differs from *V. macularis* in size of ascospores (Fig. 22i), which are 20–23 × 11–13 µm, and in conidial size and shape. Conidia (Figs. 22d–f) are never curved, and average 25–36 × 8–14 µm (Dance 1961).

Look-alikes

Cladosporium species, primarily *C. herbarum*, the asexual stage of *Mycosphaerella tassiana*, are common olive-green saprophytic molds which may superficially be confused with *Venturia* (see the section on *Septoria populicola* for a full description of the teleomorph). *Cladosporium* conidiophores (Fig. 22j) are longer and more branched, and conidia are highly variable in size, are irregularly septate, with multiple pores common (Fig. 22k).

Key Diagnostic Features

- terminal shoots and new growth on branch tips become blackened and shriveled in spring and early summer
- blackened, twisted lesions on young leaves
- velvety mats of olive green conidia on leaf spots and shepherd's crooks
- stunted, malformed understorey trees with thin foliage

Impact

Heavy levels of disease in hybrid T×D plantations on Vancouver Island (Fig. 22c) and the lower Columbia River have necessitated the replacement of susceptible clones by more resistant ones (Newcombe and Van Oosten 1997). Repeated blighting for several years results in stunted malformed trees. In Italy, *V. populina* causes losses of approximately 30% in growth of susceptible trees (Giorcelli and Vietto 1992). Spring defoliation increases the competitive advantage of weeds by allowing greater light penetration (Newcombe and van Oosten 1997). This potentially increases vegetation management costs in the first years of establishment.

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Other Foliar Diseases

Phaeoramularia maculicola (Rom. & Sacc.) Sutton (= *Cladosporium sessile* Ell. & Barth.) (Hyphomycetes)

This fungus is associated with a minor leaf spot disease of aspen and black cottonwood in B.C. It causes variable degrees of annual foliar damage to native aspens and cottonwoods throughout North America, and *P. tremula* in Europe. Phaeoramularia leaf spots are, for unknown reasons, most commonly seen on young trees on sand bars. Foliar lesions (Fig. 23a) are grayish brown with a darker border, circular, 3–10 mm in diameter. The fungus develops in dark tufts in the center of the spots on the undersurface of the leaves, frequently protruding conidiophores through stomates (Fig. 23b). Conidiophores are unbranched, pale brown, 1-septate at the base, with clusters of old conidial secession scars at the apex. Conidia (Fig. 23c) are pale yellowish brown, 0–1 (rarely 2 or 3) septate, oblong to fusoid, smooth-walled, and measure 12–15 × 3 µm (Sutton 1969).

Symptoms caused by *P. maculicola* look similar to those caused by *Pollaccia borealis* on aspen (described elsewhere). However *Pollaccia* usually sporulates on the upper surfaces of the leaves, and produce much shorter conidiophores without the distinctive conidial secessional scars.

Figs. 23a–c. *Phaeoramularia maculicola* on black cottonwood.

Fig. 23a. Leaf spots on young cottonwood.

Fig. 23b. Conidiophores (stained in cotton blue) emerging from stomates. Note prominent apical conidial secession scars. Photomicrograph (× 1000).

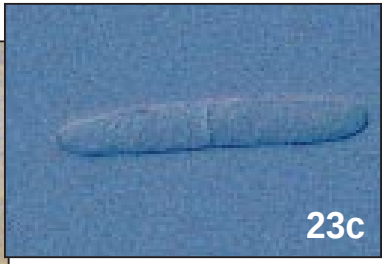

Fig. 23c. Conidium. Photomicrograph (× 2000).

Septotinia podophyllina (Whetzel) Groves and M.E. Elliott (Ascomycetes, Helotiales) Anamorph = *Septotis podophyllina* (Ellis and Everh.) Arx (not illustrated)

Known primarily from eastern North America on *P. deltoides*, the anamorph of *Septotinia podophyllina* has also been recently collected from blighted T×D hybrid poplars in plantations on Vancouver Island. This fungus is also pathogenic to *Salix*, *Prunus*, and *Podophyllum*. Affected leaves, which occur at low levels on susceptible poplar trees, are almost completely browned. The infections, which expand in striking, pale brown to greyish concentric rings, require wounds (such as insect feeding damage) in order to become established. Whitish to cream-colored clusters of conidia and conidiophores (sporodochia) are produced in large numbers on the leaf surface. Conidia are hyaline, cylindrical, slightly pointed at the apical end, 0–4 septate, and measure 17–40 × 5–7 µm. Towards the end of the growing season, small (<1 mm in diameter) dark sclerotia are produced in dead leaf tissue (de Kam 1973). The sclerotia overwinter on fallen dead leaves, but the sexual (*Septotinia*) stage (apothecia which develop on sclerotia in the spring) has not yet been documented in B.C.

Powdery mildew

Uncinula salicis (DC) Winter (Ascomycetes, Erysiphales)

Powdery mildew is occasionally encountered on suppressed understorey leaves of cottonwood and hybrid poplar, especially under greenhouse conditions. *Uncinula salicis* is also pathogenic to *Salix* spp. Dusty to cottony white clumps of superficial hyphae and conidia (Fig. 23d) may engulf the entire upper leaf surface by the end of the growing season. Large numbers of hyaline ellipsoid single-celled conidia measuring 25–30 × 15–19 µm are produced in chains on the white mycelium. Mid to late summer, the sexual state develops in cleistothecia, which appear to the naked eye as small orange to black globes. Microscopically, the cleistothecia bear an equatorial fringe of hyaline appendages with coiled tips (Fig. 23e), and contain 8–12 pear-shaped yellowish to hyaline asci. Asci (Fig. 23f) bear 4–5 broadly ellipsoid hyaline single-celled ascospores measuring 25–30 × 11–20 µm (Funk 1985).

Fig. 23d–f. *Uncinula salicis* on cottonwood.

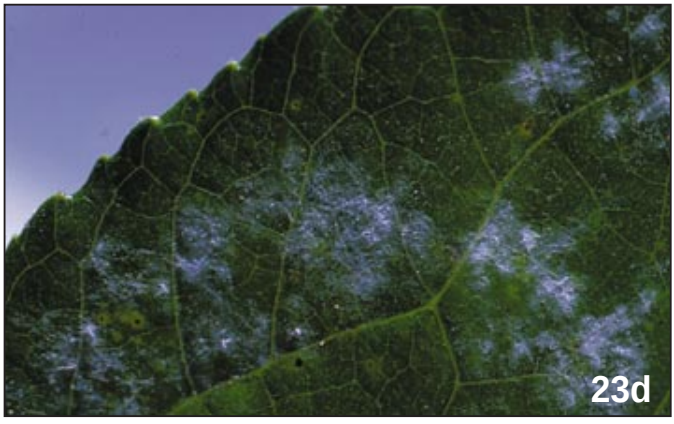

Fig. 23d. White mats of hyphae and conidia on leaf surface.

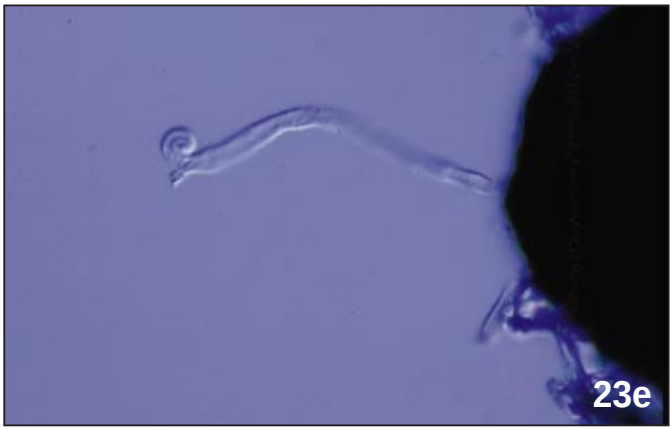

Fig. 23e. Coiled tip of a cleistothecial appendage. Photomicrograph (× 300).

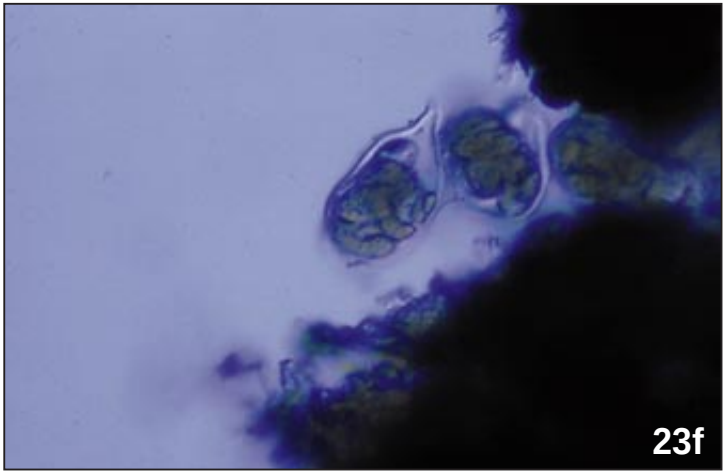

Fig. 23f. Asci. Photomicrograph (× 160).

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Most wood decay fungi on poplars cause a white rot, breaking down both cellulose and lignin components of the wood. Advanced decay columns are often softened to the point that the remaining tissues are easily pulled apart by hand. Over years, decay of a fallen tree usually involves a succession of fungal populations, each able to degrade the wood left behind by its predecessor.

Lindsey and Gilbertson (1978) documented over 250 species of fungi associated with aspen decay in North America, and also provided an excellent review of North American decay studies. Decay organism populations tend to be host specific, and these numbers do not necessarily reflect the biodiversity of decay fungi on other poplar species. In B.C., there are 148 documented records of basidiomycetous poplar decay fungi (see Host-Fungus Index). Of these records, 28 species have been found only on *P. tremuloides*, and 76 have been found only on *P. trichocarpa*. Forty-four other species are common to both hosts. Several ascomycete taxa are also associated with wood decay (*Cryptosphaeria* and *Entoleuca*, for example) but they are discussed herein with their associated canker diseases. Thomas et al. (1960) found 17 species of fungi causing decay in aspen and balsam poplar in boreal Alberta. Only five of these fungal species were common on both hosts.

The species which are able to colonize and decay heartwood in standing, live trees are most economically damaging, and play important roles in devaluing stands of merchantable poplar. For example, the pathological rotation age of an aspen stand (age after which growth increment is less than loss due to decay) is as low as 40–50 years in regions such as Minnesota, where decay caused by *Phellinus tremulae* is the limiting factor. However, this age appears to be quite variable depending upon the region studied. For example, Basham and Morawski (1964) concluded that aspen in Ontario should be harvested before 80 years in order to avoid heavy losses due to decay. Data from B.C. indicate that the appropriate rotation age for aspen ranges from 80–90 years on poor sites to 60–70 years on medium and good sites, not taking into account levels of decay (Peterson and Peterson 1992). An early study on decay in cottonwood stands in B.C. (Thomas and Podmore 1953) identified 70 species of fungi causing decay, only a few of which caused significant losses in living trees. Ninety-two percent of the losses were attributed to *Spongipellis delectans* and *Pholiota populnea*. Forty-five years later, the impact of these two fungi elsewhere in the province is still more or less unknown. *Bjerkandera adusta* and *Armillaria mellea* (*sensu lato*) were also associated with significant volume losses in living tress, while *Ganoderma applanatum* contributed significantly to butt decay losses in dead trees.

Intensively managed short-rotation (7–15 years) hybrid poplar plantations are likely to remain unaffected by heart rot organisms, which tend to become established on older trees.

The most common and damaging fungal species on standing live poplars in B.C. are described and discussed, along with a few common saprophytes.

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Key to Common Non-gilled Wood Decay Fungi

1. Undersurface of fruiting body covered with pores 3
1. Undersurface/spore bearing surface of fruiting body not poroid 2

   2. Undersurface of fruiting body covered with spines . . . . . .*Radulodon americanus*
   2. Spore-bearing surface smooth; fruiting bodies attached directly to bark, or curling away only at edges, pigmented reddish to purplish when fresh . . . . .*Peniophora* and related taxa

3. Fruiting body with consistently rounded to slightly angular pores 4
3. Fruiting body with some maze-like pores . . . . . .*Daedaleopsis confragosa*

   4. Fruiting body perennial (woody, and with more than one layer of pores when sectioned vertically) 5
   4. Fruiting body annual (fleshy or leathery, with only one pore layer) 6

5. Fruiting body usually appearing at a branch stubs of standing trees, hoof-shaped; undersurface pale brown with pores lined with pointed setae, and producing hyaline basidiospores with simple cell walls . . . . . .*Phellinus tremulae* on aspen; *P. igniarius* on other poplar species
5. Fruiting body bracket-shaped, usually appearing at or near the ground or on fallen trees. Fresh pore surface pale, darkening when scratched. Pores without setae, producing reddish brown double-walled basidiospores . . . . . .*Ganoderma applanatum*

   6. Fruiting body with a pale tan or white pore layer 7
   6. Fruiting body with a brightly pigmented or dark pore layer 8

7. Fruiting body large, usually singular, up to 15 cm wide × 4 cm thick, pale, fleshy to leathery, with large pores averaging 1–2 per mm, exclusively on black cottonwood . . . . . .*Spongipellis delectans*
7. Fruiting body thin, leathery, usually clustered in large numbers, with small pores averaging 5–6 per mm, on aspen and black cottonwood . . . . . .*Trametes versicolor*

   8. Pore layer dark smoky grey . . . . . .*Bjerkandera adusta*
   8. Pore layer violet colored, fading to buff . . . . . .*Trichaptum subchartaceum*

Key to Four Common Mushrooms Associated with Decay

1. Mature mushroom producing a white spore print* 2
1. Mature mushroom producing a dark spore print 3

    2. Brown mushrooms with central stipes, produced in clusters at bases of dead or dying trees, associated with white mycelial fans under the bark, and black, string-like rhizomorphs . . . . . .*Armillaria* spp.
    2. White to pale tan mushrooms with laterally attached stipes, emerging from bark on trunks of fallen, standing dead or topped trees . . . . . .*Pleurotus* spp.

3. Large clusters of mushrooms with gills that dissolve to produce an inky black fluid, on roots with brown cubical decay . . . . . .*Coprinus* spp.
3. Small clusters or single large fleshy mushrooms producing a brown spore print, fruiting on cut ends of cottonwood logs, associated with a yellowish laminated heart rot . . . . . .*Pholiota populnea*

* Techniques for collecting mushrooms and obtaining spore prints are described in the *Field and Laboratory Techniques* section.

Armillaria Root Diseases

Causal Agents

Armillaria sinapina Bérubé and Dessureault (Basidiomycetes, Agaricales)
A. nabsnona Volk and Burdsall (formerly known as NABS IX)
A. ostoyae (Romagnesi) Herink

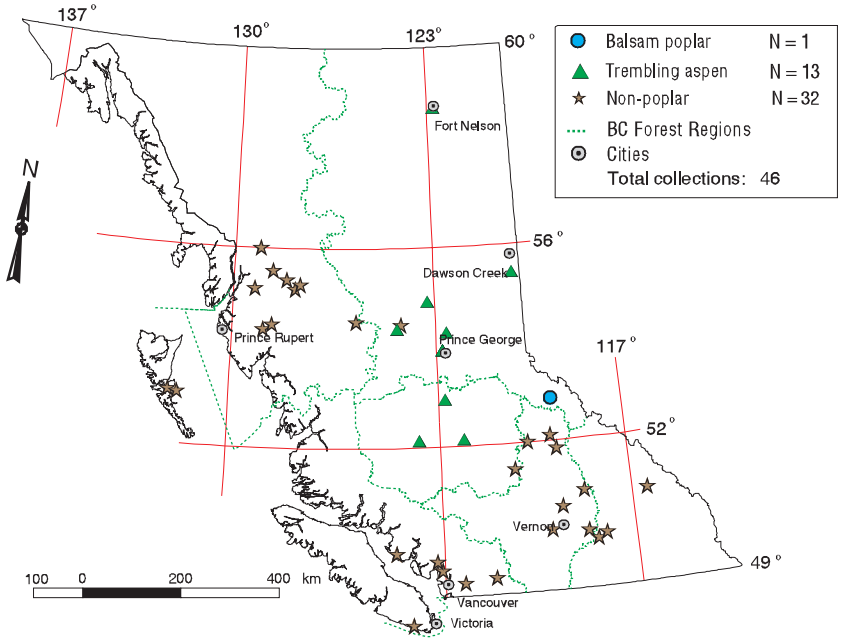
Older literature references (prior to the late 1970s) frequently use one name, *A. mellea* (Vahl:Fr.) P. Kumm. in reference to all three of these species. The old species complex of *A. mellea* in North America has now been split into nine distinct biological species (Anderson and Ullrich 1979; Morrison et al. 1985; Anderson 1986).

Hosts/Host Specificity

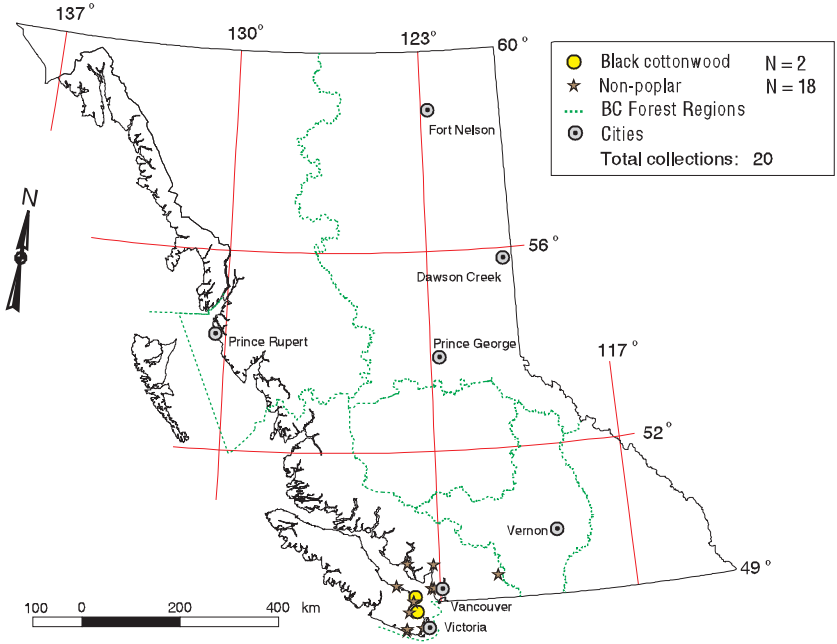
Incidence and host specificity of *Armillaria ostoyae* have been well documented on conifers in B.C., hence the extensive distribution map for *A. ostoyae* on all hosts. This species, which is principally pathogenic to living conifers, occurs south of a line drawn between McBride, Williams Lake, and Bella Coola, and is widespread from 49° to 53°N (Morrison et al. 1985). *Armillaria ostoyae* has been reported infrequently on *Populus tremuloides* in south-central B.C.; further collections and accurate species identifications are required to determine the role that this fungus plays in hardwood stands. Anecdotal evidence suggests that an association exists between stressed poplar trees and *Armillaria* infection.

Armillaria sinapina has been documented on dead or dying *P. tremuloides* and *P. trichocarpa* as well as other hardwoods, and as a secondary invader of conifers killed by other agents. It is also associated with stringy butt rot on these hosts, and is widespread throughout the province from 49° to 57°27'N, overlapping in distribution with the entire range of *A. ostoyae* (Morrison et al. 1985).

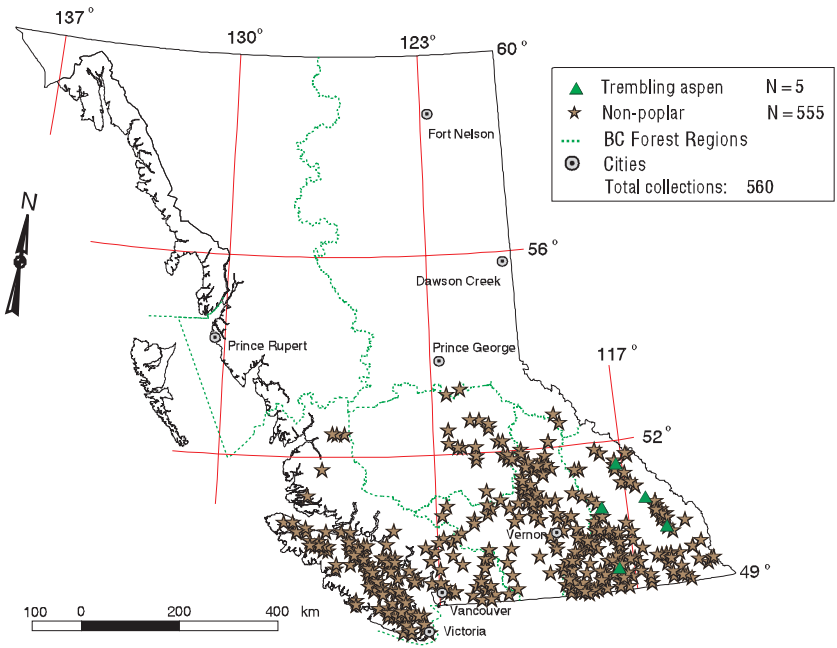
Distribution of *Armillaria sinapina.*



Distribution of *Armillaria nabsnona*.



Distribution of *Armillaria ostoyae*.



Armillaria nabsnona, currently known only from the southwest in B.C., has been verified on *P. trichocarpa* on Vancouver Island, where at one site near Duncan it was associated with butt rot and windthrow of mature trees. It is also found in Alaska, Idaho, and California (Volk et al. 1996) which suggests that further study might extend the known distribution in B.C. This species has been most frequently collected on *Alnus* in riparian areas, and has also been collected from *Acer* in B.C. and elsewhere.

Further collections of all three of these species from hardwoods will be required before the host-fungus relationships of *Armillaria* on *Populus* in B.C. is better understood.

Time of Appearance

Perennial infections may be detected any time of the year on the trunk or butt, but crown symptoms are best detected during the growing season. Fruiting bodies (mushrooms) are produced in late summer to fall at the base of dead or infected trees and stumps, where wood is exposed (usually at ground level).

Source of Inoculum

Colonized wood is the primary source of inoculum. Root-like *Armillaria* rhizomorphs grow through the soil from woody food bases such as roots, or stumps, until they reach and infect a susceptible host. The fungus may also spread via root contact in dense stands. The role of basidiospores in dispersal of the pathogen is not well understood.

Description

Root disease centers are generally small, but may be detected by the characteristic pattern of dead, fallen trees surrounded by dying trees. However, root disease may also occur in a more scattered pattern on individual trees. Crown symptoms indicating root disease include premature foliage loss, chlorosis, and reduction in shoot growth.

Armillaria mushrooms are ephemeral, and in some years rare; therefore they and their morphological features are rarely used for routine disease diagnosis and species determination. Briefly, the mushrooms (Figs. 24e, f), which often occur in large clusters on stumps and exposed dead wood of living trees, are tan to honey to reddish-brown, with caps 5–12 cm in diameter, and stalks with an annulus (collar) on the upper half. Gills, which are cream to pale brown, discharge masses of white to pale cream basidiospores, which often collect on the caps of adjacent mushrooms in the cluster. Detailed descriptions of fruiting bodies of each of the above species are found in papers by Bérubé and Dessureault (1987 – *A. sinapina*), Volk et al. (1996 – *A. nabsnona*) and Shaw and Kile (1991 – *A. ostoyae* and other species).

Species are more commonly identified from culture morphology, mating studies, or genetic analysis (Shaw and Kile 1991). In the field, identification of Armillaria root disease is fairly straightforward. The bases of infected trees often become blackened, with longitudinal cracks and fissures at and above ground level on the butt of the tree. Lesions up to 1 m in length may also develop at the bases of trees with advanced infections, exposing the wood beneath as dead bark sloughs off. When the bark at the bases of diseased trees is cut away, whitish plumes of fungal tissue, called mycelial fans, are evident. In aspen, (Fig. 24a) the fans may be extensive and prominent, extending over 1 m above ground line on standing dead and dying trees. In cottonwood, (Figs. 24c, d) mycelial fans are thinner, less extensive, and often difficult to separate from the convoluted, thick bark found at the bases of large, diseased trees. Black, strap-like tough, flexible fungal strands called rhizomorphs (Fig. 24f) may be found on root surfaces, in adjacent soil and on hollow butts of trees with advanced decay. Rhizomorphs range from fine, 1 mm-wide threads to 1 cm-wide leathery straps, and may be over 1 m in length. Wood decayed by *Armillaria* spp. (Fig. 24b) is yellow, stringy, and often surrounded by dark brown fungal and wood material (Hiratsuka et al. 1990).

Fig. 24. Armillaria root disease of aspen and cottonwood.

Fig. 24a. Mycelial fans of *Armillaria sinapina* on dying trembling aspen (bark has been removed).

Fig. 24b. Butt rot of overmature trembling aspen.

Figs. 24c, d. Mycelial fans (either *A. sinapina* or *A. nabsnona*) on black cottonwood.

Fig. 24e. Mushrooms clustered on downed trees in the fall.

Fig. 24f. Excavated mushroom, with rhizomorphs attached to the base of the stem.

Key Diagnostic Features

- white mycelial fans, advancing 1 m or more under the bark of aspen with advanced infections. Fans on cottonwood are less obvious or extensive but are still evident at the very base and on the roots of infected trees.
- yellow-white stringy butt and root rot. Advanced decay is very spongy and soggy.
- black shoestring-like rhizomorphs on root surfaces and in adjacent soil. Rhizomorphs of *A. ostoyae* are fewer and dichotomously branched (forking in pairs at an acute angle) while the rhizomorphs of *A. sinapina* are more numerous and monopodially branched (at right angles from a common axis). Rhizomorphs may be rare or lacking altogether in trees infected with *A. nabsnona,* but when they are present they are indistinguishable from those of *A. sinapina* (Morrison et al. 1992).
- clusters of tan to honey-brown mushrooms at the base of trees and stumps. Mushrooms produce a white spore print, and are often attached to rhizomorphs.

Look-alikes

Other brown mushrooms may be found fruiting on downed poplar trees. *Pholiota* and *Coprinus* spp. are commonly encountered but these mushrooms produce a black spore print. *Flammulina velutipes* (Curt.:Fr.) Singer is a clustered orange-brown mushroom with white spores and minutely hairy stalks. It has been occasionally isolated from cottonwood (Thomas and Podmore 1953) and aspen in B.C., and is known to be associated with a white butt rot of aspen elsewhere in North America (Hinds 1985).

Impact

Although infection levels in poplar stands in B.C. are not known, *Armillaria* infections elswhere in North America are known to decrease height and diameter growth, as well as cause root and bole decay, and tree mortality, limiting rotation length and the number of times aspen stands can be successfully vegetatively regenerated. Stanosz and Patton (1987) demonstrated that root rot caused by *Armillaria* species significantly decreased levels of suckering after the third short rotation of aspen stands in Minnesota and Ontario. It is possible that *Armillaria* root disease could play a role in decline of aspen stands after repeated harvesting in B.C. as well.

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White Mottled Trunk and Butt Rot

Causal Agent

Ganoderma applanatum (Pers.) Pat. (Basidiomycetes, Aphyllophorales)

Hosts/Host Specificity

Ganoderma applanatum is associated with heart and butt rot of *P. tremuloides*, *P. trichocarpa* and *P. balsamifera* in B.C. In this province, it is also associated with stem and butt decay of a wide variety of hardwood (including *Acer*, *Alnus*, *Betula*, *Salix*, and *Quercus*) and conifer hosts. It is widespread throughout the northern hemisphere as far south as India.

Time of Appearance

Decay columns are evident in recently windthrown trees even before conks develop. Conks, which are perennial, are more commonly encountered on dead trees on damp sites.

Source of Inoculum

Basidiospores, which are wind-borne and produced annually by the trillions in large perennial conks, are the main source of inoculum.

Description

Conks most commonly appear low on the butt of dead standing trees, or near the ground (often with grasses or other plants engulfed by growth of the fungus) on fallen trees. They also grow on the cut end of logs and exposed roots. Incidence appears to be more common on moist sites and in mature stands (Ross 1976). Conks rarely occur on standing live trees, but large decay columns present on recently dead trees indicate that the fungus was established long before tree death.

Conks are whitish and round the first year that they protrude from the tree, but in subsequent years they become flattened, semicircular, and shelf-like, 5–80 cm across and 2–20 cm thick. The upper surface is tan to brown, concentrically ridged, often lumpy, and dusted with a reddish-brown layer of basidiospores (Fig. 25a). The lower surface is whitish to yellowish-tan, covered with tiny pores, 4–6 per mm. If scratched or handled the pore layer turns reddish-brown. The damaged pore surface will remain permanently stained if the conk is allowed to dry (Fig. 25b). This feature has resulted in the common name of "artist's conk" for this fungus.

Internally, the conk is dark chocolate brown, and concentrically ringed with annual pore layers which may be used to age it. The diagnostic microscopic feature of this fungus is the distinctive basidiospores, which are reddish-brown, darkening in Melzer's reagent (dextrinoid), ovoid, slightly flattened and bearing a pore at the distal end (Fig. 25d), 8–12 × 8–10 µm, with two distinct wall layers separated by small pegs which appear as barely visible dots under high magnification (Fig. 25c).

Incipient wood decay first appears as bleached areas, with numerous brown to black zone lines, the wood becomes white and mottled, and finally yellowish-white, soft, and stringy (Gilbertson and Ryvarden 1986).

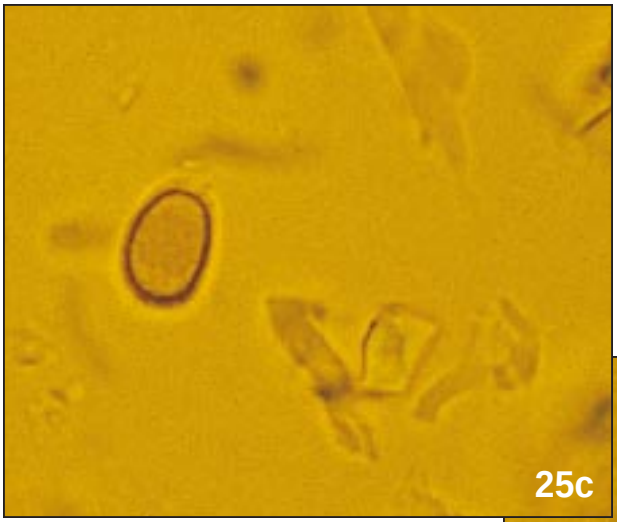
Key Diagnostic Features

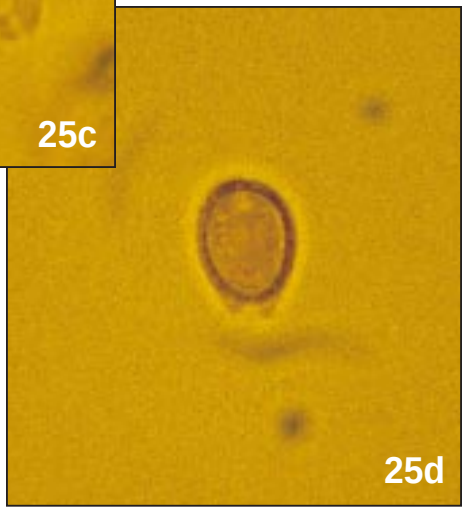
- dark brown, flat, shelf-like "artist's" conks, with reddish brown dusting of spores on upper surface, and a whitish poroid undersurface, which stains brown where handled
- characteristic ornamented, dextrinoid basidiospores

Fig. 25. *Ganoderma applanatum* – white mottled trunk and butt rot.

Fig. 25a. Mature shelf-like conks, covered and surrounded by a reddish brown spore deposit.

Fig. 25b. Conk on fallen trembling aspen; brown stain on bruised pore surface evident at right.





Figs. 25c, d. Basidiospores, stained with Melzer's reagent, showing wall ornamentation (Fig. c) and pore at end of spore (Fig. d). Photomicrographs (× 1100; × 1250).

Impact

Aspen windthrow due to Ganoderma butt and root rot appears to be the main factor limiting tree size in mature stands in Wyoming, as well as other areas in the Rocky Mountains (Ross 1976). Some 86% of the blowdown in a Colorado aspen stand was attributed to this fungus, which caused an estimated volume loss equal to that caused by *Phellinus tremulae* (Landis and Evans 1974). Similar relationships have not yet been documented on aspen in B.C., where the impact of this fungus is relatively unknown. On cottonwood near Quesnel, B.C. (Thomas and Podmore 1953), *G. applanatum* was third in relative importance of the decay fungi, and third in total decay volume after *Spongipellis* (= *Polyporus*) *delectans* (Peck) Murrill and *Pholiota populnea* (Pers.: Fr.) Kuyper & Tjall-Beukers (= *P. destruens* (Brond.) Gill.).

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Peniophora Decay

Causal Agents

Peniophora polygonia (Pers.:Fr.) Boud. & Galzin (Basidiomycetes, Aphyllophorales)
Peniophora rufa (Fr.:Fr.) Boidin

Hosts/Host Specificity

Both fungi have been reported on all native and hybrid poplar hosts in B.C. The most frequently documented host-fungus association in the province is between *Peniophora polygonia* and *Populus tremuloides*. *Peniophora rufa* is also commonly associated with *Populus tremula* in Europe (Chamuris and Falk 1987), and other hardwoods in North America, including *Quercus*.

Time of Appearance

Fruiting bodies are more numerous on broken or wounded branches; therefore, increased incidence could be expected after thinning operations or in areas of high activity such as recreation sites.

Source of Inoculum

The wind-borne basidiospores serve as the inoculum source.

Description

These two closely related fungi are separable from each other by fruiting body size and by microscopic features.

Peniophora polygonia occurs on dead, attached or fallen branches, branch stubs, and old exposed trunk scars. Fruiting bodies are flat, scaly, 5–10 cm wide × 1–2 mm thick, pushing through holes in bark, often having wart-like surface thickenings. The surface of the fruiting bodies is reddish with a whitish outer bloom (Fig. 26b). When fruiting body tissue is sectioned and examined microscopically, large gloeocystidia (sterile swollen hyphae) are prominent, reaching 100 × 15–25 µm, and staining purplish-black in sulfovanillin (Fig. 26c) (see Diagnostic Reagents section). The surface of the fruiting body is whitened by dendrohyphidia, characteristic branched, convoluted sterile hyphae 1–2 µm wide. Basidiospores (Fig. 26d) are cylindric, slightly curved, 9–12 × 2.5–4 µm, hyaline, but produce a pale red spore deposit if fruiting

Fig. 26. Peniophora and similar wood decay fungi of poplar.

Fig. 26a. *Peniophora rufa* on dead standing T×D hybrid.

Fig. 26b. *Peniophora polygonia* on trembling aspen.

bodies are placed facedown on white paper when fresh (Ericksson et al. 1978). Decay columns are stained reddish brown, and contain pockets of pinkish to brownish decay. Althought these columns do not usually extend to great lengths in aspen trunks, they are very common, and total areas of decay in the trunk may be significant (Hiratsuka and Loman 1984).

Small *Peniophora polygonia* fruiting bodies may be confused superficially with *Peniophora rufa* (Fig. 26a) but they may be distinguished by microscopic features. *Peniophora rufa* fruiting bodies are smaller, 3–10 mm in diameter, wart-like, red to reddish brown with a whitish surface bloom, but fading to pinkish grey when dry and overmature.

Key Diagnostic Features

- flat reddish fruiting bodies with whitish surface bloom
- large cystidia turning blackish purple in sulfovanillin
- decay column with irregular pockets of pinkish brown decay

Look-alikes

Chondrostereum purpureum (Pers.:Fr.) Pouzar: Fruiting bodies (Fig. 26e) have a reddish-purple fertile layer, but are often shelf-like, unlike *Peniophora* species, and the cystidia do not stain in sulfovanillin. Basidiospores are cylindrical, 6–8 × 2.5–3 µm (Fig. 26f). This fungus is associated with white rot of many hardwood species, including *Populus*, and is currently being developed as a mycoherbicide at the Pacific Forestry Centre. It is intended for use in control of *Alnus rubra* Bong., and is only weakly pathogenic to poplar (Wall 1996).

Punctularia strigoso-zonata (Schwein.) Talbot: The surface of this fungus (Fig. 26g) is reddish-purple, zonate, and wrinkled, with surface folds in radiating patterns. The hymenium bears dendrohyphidia, but no cystidia, unlike *Peniophora* species. Basidiospores are broadly cylindrical to ellipsoid, 7.5–9 × 3–4 µm. This fungus is associated with white rot of aspen and cottonwood logs and slash.

Radulodon casearium (Morg.) Ryvarden produces a completely different (toothed, white) type of fruiting body, but the type of wood decay and stain produced by this fungus is nearly identical to that produced by *Peniophora polygonia* (Hiratsuka and Loman 1984).

Impact

According to Basham (1958), *Peniophora polygonia* functions as a pioneer decay fungus in newly dead wood. During colonization, *Peniophora polygonia* alters the structure of host wood such that other decay fungi, such as *Phellinus tremulae* and *Ganoderma applanatum*, can become established. *Peniphora rufa* was found to be the second most common species fruiting on dead aspen limbs in an aspen plantation in central New York (Chamuris and Falk 1987).

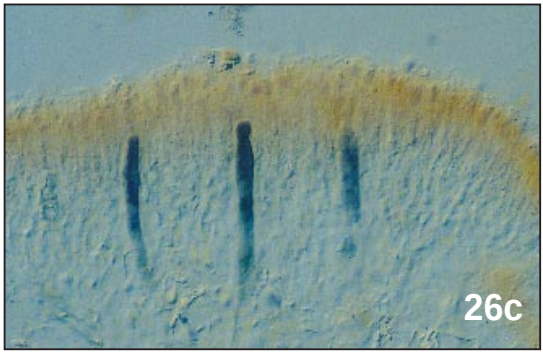

Fig. 26c. Gloeocystidia of *Peniophora polygonia*, stained black in sulfovanillin. Photomicrograph (× 100).

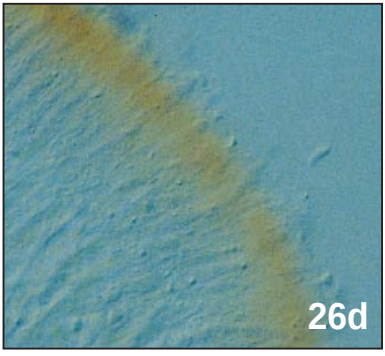

Fig. 26d. Basidiospores of *P. polygonia*. Photomicrograph (× 100).

Fig. 26e. *Chondrostereum purpureum* on trembling aspen.

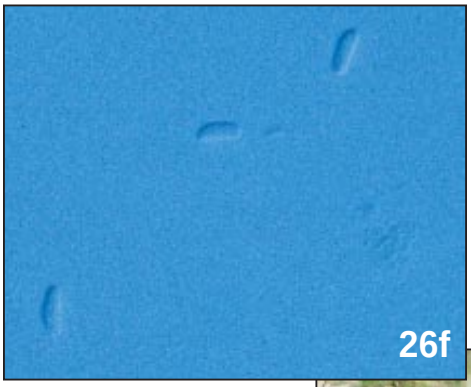

Fig. 26f. Basidiospores of *C. purpureum*. Photomicrograph (× 850).

Fig. 26g. *Punctularia strigoso-zonata* on trembling aspen.

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Aspen Trunk Rot

Causal Agent

Phellinus tremulae (Bondartsev) Bondartsev & Borisov (Basidiomycetes, Aphyllophorales)
Synonym = *Phellinus igniarius* (L.:Fr.) Quél. var. *populinus*
Synonym = *Fomes igniarius* (L.:Fr.) J. Kickx fil. *f. tremulae* Bondartsev

Hosts/Host Specificity

In B.C., *Phellinus tremulae* occurs exclusively on *Populus tremuloides*. In the rest of North America, *P. tremuloides* and *P. grandidentata* are the sole hosts. In Europe, it has also been reported from *P. tremula* L., *P. alba* L., and *P. x canescens* (hybrid between *P. deltoides* × *P. nigra*) (Niemelä 1974).

Time of Appearance

Fruiting bodies or blind conks (sterile masses of fungal tissue) may not appear for many years after the initial colonization of heartwood from a branch stub or wound. However, no other external indicator of decay is visible.

Source of Inoculum

Wind-borne basidiospores dropped from the pores of mature conks serve as primary inoculum of branch stubs and other wounds where heartwood is exposed.

Description

Conks (Fig. 27a) are perennial, producing a new pore layer each year, and woody. They usually develop at branch scars, reaching sizes up to 20 cm wide × 15 cm thick, and are triangular in section when split in half parallel to the long axis of the tree trunk. The inner context of the conk is striped with thin, faint vertical lines due to white fungal tissue stuffing the old pore

Distribution of *Phellinus tremulae*.

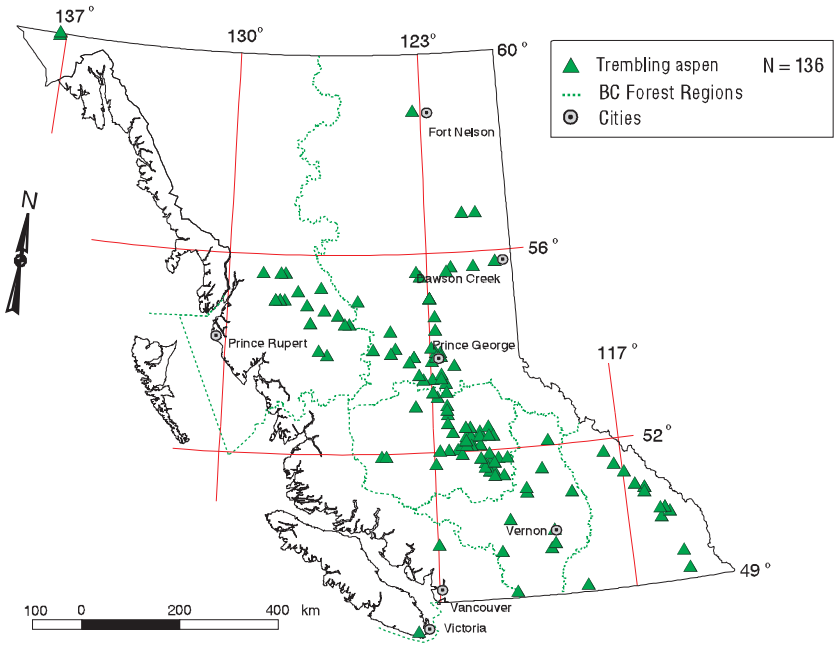

Fig. 27. *Phellinus tremulae* on trembling aspen.

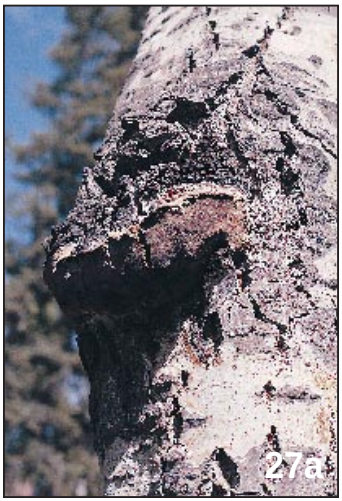

Fig. 27a. Mature hoof-shaped conk on a standing live aspen.

Fig. 27b. Aspen trunk cut below conk, showing white heart rot and black zone lines.

Fig. 27c. Brown pore layer of mature conk.

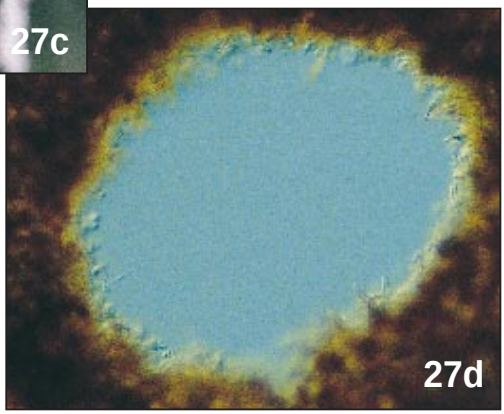

Fig. 27d. Cross-section of pore, showing setae and globose hyaline basidiospores. Photomicrograph (× 100).

Fig. 27e. Black gall, with *P. tremulae* conk adjacent to it (see also Fig. 3d).

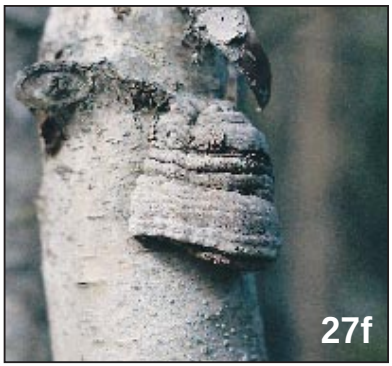

Fig. 27f. *Fomes fomentarius* conk, common on other hardwoods, and occasionally occurring on *Populus* spp.

layers. The pore surface (Fig. 27c), which is a purplish brown, angles at approximately 45° from the outer edge of the conk to the lower edge where it touches the bark of the tree. A hand lens or dissecting microscope reveals the fine pores which occur at a density of about 5–7 per mm. The pores (Fig. 27d) are lined with pointed, thick-walled setae, 12–30 × 6.0–7.5 µm, which turn brown in KOH. Hyphae on the end-walls of the pores are vertically parallel, an important taxonomic feature separating *Phellinus tremulae* from closely related species.

Basidiospores are hyaline, subglobose to broadly ellipsoid, thick-walled, averaging 4.5–5(-6) × 4–5 µm (Gilbertson and Ryvarden 1987).

Phellinus tremulae causes a white heart rot (Fig. 27b) which is characterized by numerous conspicuous black zone lines surrounding each decay column. In cross section these lines are 1 mm or less in thickness. Within the zone lines the wood is soft and white. When dry, wood with advanced decay may be broken by a fingernail. Outside of the zone lines, wood is often stained reddish brown in areas of incipient decay. Decaying wood has a distinctive sweet wintergreen odor (Hiratsuka et al. 1990; Hiratsuka and Loman 1984).

Look-alikes

If a conk has been separated from the tunk of the tree, and the host is unknown, *Phellinus tremulae* may be very difficult to distinguish from the remainder of the closely related *Phellinus igniarius* complex (Niemelä 1975). After proper host identification and microscopic examination, the two groups of fungi are easily distinguished from each other. Setae of *P. igniarius* (L.: Fr.) Quél. are smaller, averaging 14–17 × 4–6 µm, and the conks tend to be more hoof-shaped, with the pore surface closer to right angles to the trunk (Niemelä 1974; Gilbertson and Ryvarden 1987).

Phellinus igniarius occurs on hardwoods other than aspen — predominatly *Alnus*, *Betula*, and *Salix*, – but also rarely on *Populus trichocarpa* (Thomas and Podmore 1953). *Fomes fomentarius* (L.: Fr.) Kickx also occurs on hardwoods (rarely on poplar) in B.C. (Fig. 27f) but is distinctive in its paler conks, pale brown pure layer and pale brown interior, and large basidiospores averaging 12–18 × 4–7 µm (Gilbertson and Ryvarden 1986).

Key Diagnostic Features

- woody hoof-shaped perennial conks on aspen
- reddish stain surrounding decay column
- wintergreen odor of decaying wood

Impact

Phellinus tremulae occurs throughout the natural range of trembling aspen on mainland B.C., but has rarely been documented from aspen on Vancouver Island. Most occurences have been recorded from the central Cariboo Region and the southern Prince George Region.

In a recent field survey of wood decay of various aspen stands in B.C., the most common decay fungus detected was *P. tremulae* (Wood and Van Sickle 1989). An average of 89% of the trees and 28% of the wood in 21 aspen stands surveyed in the central and northern interior of the province were either decayed or discolored. In the Prince George Region, only 13% of the aspen were free of stain and decay at 20 sites from west of Prince George to north of Fort St. James. Stain was present in 70%, and decay in 57% of the affected trees. At 11 sites in the Cariboo Region, where aspen grows on 3% of the forested area, 74% were discolored and 60% were decayed.

Protecting aspen from stem wounds and fire damage and maintaining dense stands may reduce incidence of decay, but levels of stain and decay in established mature stands ready for harvesting cannot be predicted reliably. Aspen affected by "black galls" appear to have reduced levels of aspen trunk rot (see section on mite stem and branch galls). However, black galls do not always prevent decay (Fig. 27c).

Although stain and decay due to *Phellinus tremulae* greatly reduce the value and yield of aspen wood in B.C., the fungus plays an important and necessary role in hardwood ecosystems. Wildlife biologists (Keisker 1987) have shown that higher incidence of wood decay, as evidenced by conks and scars, was directly related to greater populations of cavity-nesting birds. Virtually all species of primary cavity-nesting birds require trees with heart rot in order to excavate their nests.

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Other Decay Organisms

The following brief descriptions and illustrations list some of the more common wood decay fungi associated with *Populus* in B.C. Some, notably *Spongipellis delectans*, *Pholiota populnea*, *Bjerkandera adusta*, and, surprisingly, *Coprinus* spp. (shaggy manes) might, after further study, prove to be significant agents of decay and volume loss in mature stands throughout B.C. However, information on the incidence and distribution of most of these fungi is too incomplete to draw conclusions now. A detailed list of fungi associated with aspen in North America, complete with keys, has been published by Lindsey and Gilbertson (1978). Taxonomic descriptions of Aphyllophorales follow this work and those of Gilbertson and Ryvarden (1986, 1987). Taxonomic descriptions of Agaricales follow Lindsey and Gilbertson (1978) and Arora (1986).

Decay fungus: *Bjerkandera adusta* (Willd.:Fr.) P. Karst. (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: All native *Populus* spp.
Fruiting bodies: Annual, thin, and bracket-like polypore in dense imbricate clusters (Fig. 28a). The upper surface is pale tan to cream while the pore layer underneath is smokey grey (Fig. 28b). Pores are small (6–7 per mm). Basidiospores are hyaline, cylindrical, 5–6 × 2.5–3.5 µm.
Type of decay: White rot of dead hardwoods and occasionally conifers.
Remarks: This fungus caused significant volume loss in mature cottonwood studied near Quesnel, B.C. (Thomas and Podmore 1953). It is commonly associated with butt rot of aspen killed by other pathogens and pests.

Decay fungus: *Coprinus* spp. (Basidiomycetes, Agaricales)
Hosts in B.C.: *P. trichocarpa*
Fruiting bodies: Based upon description of *C. atramentarius* (Bull.:Fr.) Fr. Clustered, fragile grayish to brownish mushrooms with conical caps, 4–10 cm tall × 4–6 cm broad (Fig. 28k). Gills deliquesce, and edge of cap rolls upward and dissolves into an inky fluid to release basidiospores. Basidiospores are ellipsoid, blackish brown with a large pore at the apex, 9–12 × 5–6 µm.
Type of decay: Brown cubical root and butt rot of cottonwood.
Remarks: This fungus was found to cause significant levels of volume loss in mature cottonwood studied near Quesnel, B.C. (Thomas and Podmore 1953). *Coprinus atramentarius* is associated with brown cubical rot of standing aspen and stumps in Wyoming (Ross 1976).

Decay fungus: *Daedaleopsis confragosa* (Bolton:Fr.) J. Schröt. (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: *P. tremuloides* and *P. trichocarpa*
Fruiting bodies: Annual bracket fungi up to 12 cm wide, with buff-colored, zonate upper surface. The lower surface is covered with variable maze-like to circular pores up to 1 mm in diameter (Fig. 28d). Basidiospores are cylindrical, slightly curved, hyaline, 9–10 × 2–2.5 µm.
Type of decay: White rot of hardwood and conifer logs and slash.

Decay fungus: *Pholiota populnea* (Pers.:Fr.) Kuyper & Tjall.-Beukers (= *P. destruens* (Brond.) Gill.) (Basidiomycetes, Agaricales)
Hosts in B.C.: *P. trichocarpa, P. nigra var. italica*
Fruiting bodies: Large, pale brown to cream, fleshy gilled mushrooms, often in clusters with the oldest mushroom on top (Fig. 28g). Caps are convex, 5–20 cm broad, covered with white scales. The gills are at first white, but turn dull brown when sporulating. Stalks are strong, 5–15 cm long and 1–3 cm thick, scaly, with a white ring. Basidiospores are cinnamon-brown with an apical pore, 7–9.5 × 4.5–5 µm.

Fig. 28. Miscellaneous wood decay fungi.

Figs. 28a, b. *Bjerkandera adusta* fruiting at the base of an aspen snag, showing upper surface (Fig. a) and dark pore surface (Fig. b).

Fig. 28c. *Trichaptum subchartaceum* on fallen aspen log, showing whitish upper surface (top), and purplish pore layer (below).

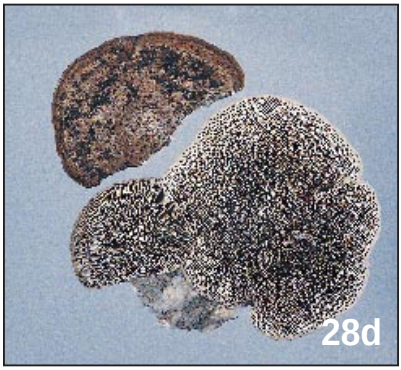


Fig. 28d. *Daedaleopsis confragosa* conks from aspen, showing gill-like to maze-like under-surface.

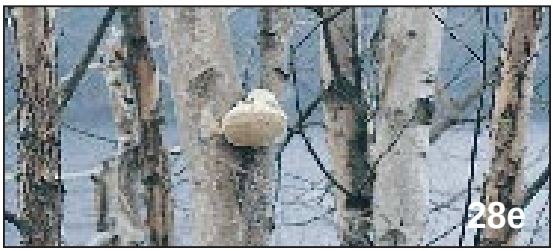


Fig. 28e. *Spongipellis delectans* fruiting on cottonwood.

Type of decay: Yellow laminated heart rot.
Remarks: Almost always seen on the cut end of large cottonwood logs. This fungus was associated with the second greatest volume loss in mature cottonwood studied near Quesnel, B.C. (Thomas and Podmore 1953).

Decay fungus: *Pleurotus ostreatus* (Jacq.:Fr.) P. Kumm. (Basidiomycetes, Agaricales)
Hosts in B.C.: *P. tremuloides* and *P. trichocarpa*
Fruiting bodies: Mushrooms with smooth, round, pale tan to grey caps up to 8 cm in diameter, and white to cream-colored gills producing cream-colored spore print (Figs. 28h, i). Some mushrooms have a short lateral to central stalk 10 mm thick × 15 mm long. Basidiospores are cylindrical, hyaline, 6–10 × 3–4 µm.
Type of decay: White rot of dead aspen and cottonwood.
Remarks: Common on dead standing and topped trees.

Decay fungus: *Radulodon americanus* Ryvarden (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: *P. tremuloides* and *P. trichocarpa*
Fruiting bodies: Annual, large, flat cream fruiting bodies covered with bristly teeth up to 3 mm long (Fig. 28j). Basidiospores are hyaline, globose, 5–6 × 4–5 µm.
Type of decay: White stringy heart rot of dead and living cottonwood and aspen; also common on *Betula*.

Decay fungus: *Spongipellis delectans* (Peck) Murrill (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: *P. trichocarpa*
Fruiting bodies: The conks are whitish, soft, irregularly hoof-shaped, fleshy, 15 cm wide × 4 cm high × 7 cm deep, breaking through bark cracks at the butt of decaying cottonwoods (Fig. 28e). Pores on the undersurface are large (1–2 per mm), circular to maze-like; basidiospores are broadly ellipsoid to subglobose, 7–9 × 5–7 µm, and hyaline.
Type of decay: White mottled rot.
Remarks: This fungus was associated with the greatest volume loss in mature cottonwood studied near Quesnel, B.C. (Thomas and Podmore 1953), but has been infrequently collected since then. The distribution records might be rare because the fruiting bodies are soft, annual, and prone to decay and insect attack.

Decay fungus: *Trametes versicolor* (L.:Fr.) Pilát (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: *P. tremuloides* and *P. trichocarpa*; widespread and common on other hardwoods and occasionally conifers.
Fruiting bodies: Annual, bracket-like, thin, clustered polypore, with the upper surface hairy and concentrically zonate in hues of brown, tan, or orange (Fig. 28f). Lower surface cream-colored, with small pores (4–5 per mm). Basidiospores are hyaline, cylindrical, slightly curved, 5–6 × 1.5–2.0 µm.
Type of decay: White rot of hardwoods and occasionally conifers.
Remarks: Common saprophyte of dead hardwoods. Often called “turkey tails.”

Fig. 28f. *Trametes versicolor* on cottonwood.

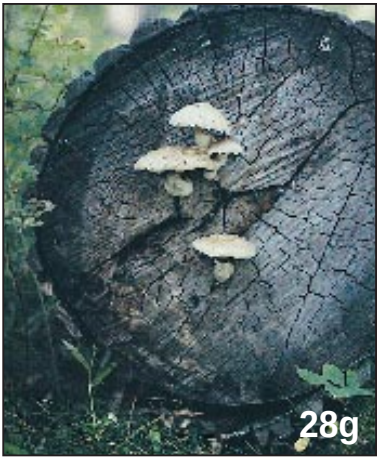
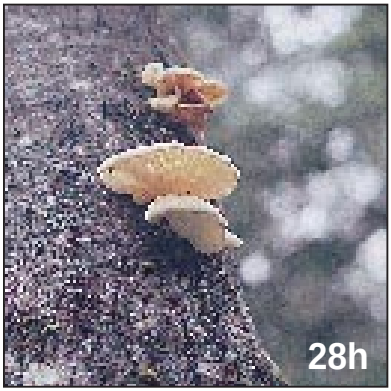


Fig. 28g. *Pholiota populnea* on cut end of a cottonwood log.



Figs. 28h, i. *Pleurotus ostreatus* on trembling aspen.

Fig. 28j. *Radulodon americanus* on trembling aspen, showing toothed pore layer.

Fig. 28k. *Coprinus atramentarius* has been associated with a brown cubical rot of aspen and cottonwood.

Decay fungus: *Trichaptum subchartaceum* (Murrill) Ryvarden (Basidiomycetes, Aphyllophorales)
Hosts in B.C.: *P. tremuloides* and *P. trichocarpa*
Fruiting bodies: Annual bracket-like clustered polypores, whose edges often fuse to form one long fruiting area over 1 m in length. Individual brackets reach sizes up to 6 cm wide and 1 cm thick, the upper surface grey to pale buff, hairy (Fig. 28c). The lower pore surface is violet-colored, fading to buff, with pores 3–4 per mm. Cystidia (sterile structures between basidia) are incrusted with hyaline crystals. Basidiospores are hyaline, cylindrical, slightly curved, 7.5–11 × 2–3 µm.
Type of decay: White pocket rot of dead, fallen aspen and cottonwood.
Remarks: Common saprophyte of dead aspen in northern B.C.

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Glossary

(adapted from Callan and Funk, 1994)*

Abaxial: referring to the lower surface of a leaf.

Abiotic pathogen: a non living cause or inducer of disease (e.g., such as a pollutant, nutrient deficiency, or adverse weather condition).

Adaxial: referring to the upper surface of a leaf.

Advanced decay: a stage of wood decay in which the wood has deteriorated in appearance and structural strength.

Aecial host: the host on which the spermagonial and aecial states of rusts develop.

Aecial state: the second spore stage in the life cycle of rust fungi producing aeciospores.

Aeciospore: rust spore formed in an aecium.

Aecium, pl. Aecia: a cup-or tube-like structure that produces chains of aeciospores.

Aerial shoot: stem-like portion of dwarf mistletoe plant outside the host bark. Its primary function is reproduction.

Alternate host: other plant species that can be host to the same pathogen. When used with respect to rusts, it usually refers to host(s) supporting different (i.e., aecial vs. telial) spore stages.

Anamorph: the asexual spore state of a fungus.

Apothecium, pl. Apothecia: cup-like structure of ascomycetes containing asci.

Ascomycetes: a biological grouping of fungi (the sac fungi) typified by the ascus within which the ascospores — typically eight — are produced.

Ascospore: sexual spore produced in an ascus.

Ascus, pl. Asci: a microscopic sac-like structure containing a definite number of ascospores, usually eight.

Asexual reproduction: reproduction not involving nuclear fusion.

Asexual state: see anamorph.

Autoecious: rusts completing the life cycle on one host (cf. heteroecious).

Bacterium, pl. Bacteria: one-celled prokaryotic microorganisms which have no chlorophyll but have cell walls, and multiply by simple division.

Basidium, pl. Basidia: a spore-bearing structure composed of one or several cells which typically produces four basidiospores externally on the surface.

Basidiomycetes: a biological grouping of fungi (mushrooms, bracket fungi, and rusts) characterized by production of sexual spores on a basidium.

Basidiospores: sexual spores produced on a basidium.

Biological control: the control of a pest by other living organisms such as viruses, fungi, bacteria or insects.

Biotic: pertaining to, caused by, or produced by living organisms.

Blight: sudden drying and browning involving whole organs such as fruits, blossoms, leaves, twigs, and shoots.

Blue sapwood stain: a deep-seated blue (sometimes more blackish or gray) discoloration confined mostly to sapwood, caused by fungi.

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* Callan, B.E.; Funk, A. 1994. Introduction to Forest Diseases. Natural Resources Canada, Canadian Forest Service, Pacific Forestry Centre, Forest Pest Leaflet 54. 16p.

Broad-sense heritability: a statistical measurement of how much a trait (i.e., disease resistance, growth form) is related to heritability rather than environmental influence. If broad-sense heritability of a particular trait is high, this indicates strong genetic control.

Brown cubical rot: a wood decay (most commonly of conifers) in which the causal fungi make a more concentrated attack on cellulose than on lignin. The brittle brown residue splits along rectangular planes in the advanced stage of decay.

Callus: host tissue that develops at the margins of wounds or cankers.

Cambium: the actively dividing layer of cells which lies between xylem and phloem tissues in higher plants.

Canker: an area of diseased tissue, often sunken, on a living stem or branch.

Canker blight: cankers that develop for one season only.

Canker rot: cankers that extend to underlying wood.

Chlorosis: yellowing of normally green tissue owing to subnormal chlorophyll content.

Conidiophore: a specialized hypha that bears conidia.

Conidium, pl. Conidia: an asexual spore usually formed on a specialized hypha (conidiophore) or in a pycnidium.

Conk: fungus fruiting bodies occurring on wood, usually large and firm-textured (i.e., a polypore).

Cystidium pl. Cystidia: a sterile cell that occurs at, and usually projects from, the surface of the hymenium of a basidiomycete.

Damping-off: disease of seedlings associated with withering and decay of roots and stem.

Decay: the process by which sound plant tissue (i.e., wood) is degraded by the action of fungi and other microorganisms.

Deuteromycetes: an artificial grouping of conidial fungi whose teleomorphs have not been found or are lacking. Most are closely related to anamorphs of ascomycetes, a few are more closely aligned to basidiomycetes.

Diffuse cankers: cankers containing little or no callus.

Dioecious: unisexual, with the male and female elements in different individuals.

Disease: the more or less prolonged interference with the normal structure and function of an organism.

Epiphytotic: a widespread and destructive outbreak of a plant disease; an epidemic in a plant population.

Eriophyoid mites: microscopic, worm-like, four-legged mites associated with galls, leaf deformity, or bronzing of foliage.

Frass: insect detritus such as chewed wood or excrement.

Fruiting body: a fungus structure specialized for producing spores. (e.g., conk, mushroom, apothecium, pycnidium.)

Fungus, pl. Fungi: a kingdom of parasitic or saprophytic eukaryotic organisms made of cellular filaments known as hyphae. Fungi feed by absorption and reproduce by forming spores.

Heart rot: decay characteristically confined to the heartwood.

Heartwood: the inner layers of wood which, in the growing tree, contain only a few living cells, and have ceased to conduct water and nutrients.

Heteroecious: requiring two unrelated host plants to complete its life cycle (cf. Autoecious).

Hibernaculum: a tent or sheath made out of a leaf or other material in which mites or other organisms hide or hibernate.

Host: a living organism harboring a parasite.

Host-alternating: requiring the production of spore states on two different host species to complete the life cycle of a heteroecious rust.

Hymenium: the spore-bearing layer of a fungus fruiting body.

Hymenomycetes: a biological grouping of basidiomycetes that produces spores from a layer of exposed basidia. These fungi are usually large and fleshy, such as mushrooms and conks.

Hyperparasite: an organism that is parasitic on another parasite.

Hyperplasia: plant tissue enlargement, such as brooming or galls, resulting from excessive cell division.

Hypertrophy: symptoms of excessive growth resulting from abnormal enlargement of plant tissue.

Hypha, pl. Hyphae: the basic filamentous vegetative cells of a fungus (cf. mycelium).

Immunity: having qualities that do not permit infection by a given pathogen.

Imperfect state: see anamorph (syn. asexual state).

Incipient decay: early stage of wood decay in which wood is invaded by the decay organism, but shows no visible symptoms of decay other than discoloration.

Infection court: the place on the host where a pathogen initiates infection.

Inoculum: infectious material of a pathogen.

Lesion: an area of diseased tissue.

Mycelial fan: a typically fan-like mass of hyphae, usually formed between bark and wood at bases of trees infected by *Armillaria*.

Mycelium, pl. Mycelia: a mass of fungus hyphae.

Necrosis: death of cells or tissues.

Necrotic symptoms: symptoms (usually discoloration) produced by the death of plant cells.

Obligate parasite: an organism that lives only on, and obtains nutrients from, living host tissue.

Oomycetes: a class of microscopic soil and water fungi that have a mobile, swimming spore stage (zoospore) and a thick-walled sexual spore (oospore).

Parasite: an organism living on or in, and obtaining its nutrients from, another living organism.

Pathogen: a living organism that can cause disease.

Perfect state: see teleomorph.

Periderm: protective layer of bark.

Perithecium, pl. Perithecia: flask-like fruiting body of ascomycetes, containing asci.

Phenology: the science of the relations between climate (seasons) and biological phenomena such as bud break and flowering.

Phloem: inner bark tissue which functions in the transport of substances produced in the leaves.

Photosynthesis: the production of nutrients in green plants from carbon dioxide and water. The energy for this process is obtained from sunlight acting on chlorophyll.

Primary inoculum: spores or tissue of a pathogen that cause the initial infection, often at the start of the growing season.

Pycnidium, pl. Pycnidia: a flask-like fruiting body, lined inside with conidiophores, and producing conidia (asexual spores).

Red heartwood stain: a pronounced reddish discoloration induced by fungi in the heartwood.

Resistance: ability of an organism to suppress or retard the activity or adverse effects of a pathogen.

Rhizomorph: a thread or cord-like structure made up of hyphae, frequently produced by *Armillaria* spp.

Saprophyte: an organism using dead organic material as food and commonly causing its decay.

Sap rot: a rot occurring in sapwood.

Sapstain: a stain that predominately affects the sapwood.

Sapwood: the outer portion of a woody stem, containing the functional xylem, living cells, and food reserves.

Sclerotium, pl. Sclerotia: a hard vegetative mass of fungus tissue resistant to unfavorable conditions.

Secondary fungus: a weak parasite or saprophyte that usually infects only predisposed, weakened, or dead hosts.

Secondary inoculum: inoculum produced during the growing season (cf. primary inoculum).

Septum, pl. Septa: the cross-wall in a hypha.

Sexual reproduction: reproduction involving the union of two nuclei.

Sexual state: see teleomorph.

Sign: visible portion of a pathogen on a diseased host, such as spores, mycelia, and fruiting bodies.

Spermagonial state: state in the life cycle of rusts in which spermatia are exuded in a sweet liquid produced from small flask-shaped fruiting bodies called spermagonia.

Spermatia: spores produced by spermagonia.

Sporangium, pl. Sporangia: an organism producing endogenous asexual spores.

Spore: a microscopic fungus propagule, commonly one-celled, but may consist of several cells. Its reproductive function is analagous to a seed.

Symptom: any reaction of a host to disease.

Systemic infection: a pathogenic infection that has spread internally through its host.

Target canker: a canker surrounded with concentric rings of callus.

Teleomorph: the sexually produced spore state of a fungus.

Telial host: the host on which the uredinial, telial, and basidial states of rusts develop.

Teliospore: the rust spore that germinates to produce basidia.

Telium, pl. Telia: fungal (rust) structure producing teliospores.

Urediniomycetes: a biological grouping of parasitic basidiomycetes (rust fungi) that often produce rusty spore masses on their host. Several spore states and host alternation also characterize these fungi.

Urediniospores: rust spores produced several times over the growing season and capable of reinfecting the same host.

Uredinium, pl. Uredinia: fungal (rust) structure producing urediniospores.

Vegetative state (of a fungus): a growing or food-absorbing mycelial state that precedes the production of spores.

Witches' broom: excessive branching of parts of tree crowns due to disease (fungi, viruses) or physiological disorder.

Xylem: wood; that is, the principal strengthening and water-conducting tissue of the stems, roots, and leaves of plants.

Zoospore: mobile swimming spore of an oomycete.

General Index

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Host-fungus Index for *Populus* in British Columbia

Fungi are separated by class. Numbers following name indicate the poplar host associations verified in British Columbia. Host codes are as follows:

1 = *Populus balsamifera*
2 = *P. tremuloides*
3 = *P. trichocarpa*
4 = *Populus* hybrids and/or unidentified species

Ascomycetes

Basidiomycetes

Tomentella calcicola (Bourd. & Galzin) M. Larsen 3
T. coerula (Bres.) Höhn. & Litsch. 3
T. ferruginea (Pers.:Fr.) Pat. 3
T. jaapii (Bres.) Bourd. & Galzin, nom. nud. 3
Tomentellastrum badium (Link) M. Larsen 3
Trametes hirsuta (Wulfen:Fr.) Quél. 3
T. ochracea (Pers.) R.L. Gilbertson & Ryvarden 2
T. pubescens (Schumach.:Fr.) Pilát 2,3
T. suaveolens (L.:Fr.) Fr. 3
T. versicolor (L.:Fr.) Pilát 2,3
Trechispora microspora (P. Karst.) Liberta 4
T. mollusca (Pers.:Fr.) Liberta 3
T. vaga (Fr.) Liberta 2
Tremella mesenterica Retz.:Fr. 2,3
Trichaptum biforme (Fr.) Ryvarden 2,3
T. subchartaceum (Murrill) Ryvarden 2,3
Tulasnella calospora (Boud.) Juel 3
Typhula sp. 3
Tyromyces galactinus (Berk.) J. Lowe 3

Coelomycetes

Colletotricum gloeosporioides (Penz.) Penz. & Sacc. in Penz. 4
Cryptosporiopsis sp. Bubák & Kab. 2,3,4
Cytospora chrysosperma (Pers.:Fr.) Fr. 1,2,3,4
C. nivea Sacc. 2,3
Cytosporina sp. 2,3
Dichomera sp. 2,3
Diplodia tumefaciens (Shear) Zalasky 2,3
Discella microsperma (G. Johnst.) Sutton 2,3
Libertella sp. 2,3
Marssonina castagnei (Desmaz. & Mont.) Magnus 2
M. populi (Lib.) Magnus 2,3,4
M. tremulae (Lib.) Kleb. 2,4
Phoma sp. 2,3
Phomopsis oblonga (Desmaz.) Traverso 3,4
Seimatosporium etheridgei Funk (TYPE) 2
Septoria populicola Peck 1,2,3,4
Sirodothis inversa (Fr.:Fr.) Sutton & Funk 2
S. populnea (Thuem.) Sutton & Funk 2,3

Hyphomycetes

Aegerita sp. 3
Alternaria alternata (Fr.:Fr.) Keissel. 3
Aspergillus sp. 3
Aureobasidium pullulans (de Bary) G. Arnaud 3
Cadophora sp. 3
Chalara sp. 2
Cladosporium herbarum (Pers.:Fr.) Link 2,3,4
C. sessile Ellis & Barth. 3