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BACTERIAL DISEASES

▶ 26.1 Brown blotch (bacterial blotch) *Fig. 26.1*

Pseudomonas tolaasii Paine

Brown blotch is the most common bacterial disease of commercial mushrooms, and it causes considerable economic losses each year through quality reduction.

Symptoms The symptoms most often observed include pale yellow areas or blotches on the cap that later turn golden yellow, yellow-brown or chocolate brown *(26.1)*. The stem (stipe) also may be affected. Occasionally, the caps will have an overall dingy

off-color with rapid deterioration and discoloration after harvest. Symptoms occur more frequently on mushrooms that remain wet for a long time and in places where they touch one another. Brown blotch symptoms can be confused with those of other diseases, such as verticillium spot.

Causal agent *Pseudomonas tolaasii* is an aerobic, Gram-negative, non- sporing rod, about 0.5 by 1 to 2 µm. It is fluorescent, oxidase-negative and arginine dihydrolase-negative. It can utilize L-arginine and L-arabinose, and does not grow at <4°C. It is white-line positive when challenged with ATCC 14340 (American Type Culture Collection, Rockville, MD). Drops of broth culture can induce browning on fresh slices of internal pileus tissue within 24 hours. Dissociation is common. The reference culture is ATCC 33618.

Disease cycle The pathogen is a natural inhabitant of the peat and lime used for casing material and can be isolated easily from compost after pasteurization. It probably survives between crops on structural surfaces, in debris, and on tools and other equipment. The bacterium can be moved readily from one crop to another on hands of pickers and on materials or equipment used in harvesting, by insects, mites and water droplets, and on mushroom spores. Once the disease is established, watering the crop will readily disperse the pathogen. Generally, disease incidence is highest in the first break. As the crop matures, there are fewer mushrooms and air circulates more freely around the crop resulting in better drying after watering and less disease.

Management

Cultural practices — Brown blotch is best managed by manipulating the growing environment. High relative humidity and surface wetness encourage the expression of symptoms. When the mushrooms stay wet longer than two to three hours, blotch can easily develop. Additional ventilation after watering will assist in drying. Maintaining a stable difference of 1 to 1.5°C between wet and dry bulb readings will lessen the chance of condensation and significantly reduce the incidence of blotch. When blotch is a problem, the crop should not be watered on consecutive days and growers should avoid watering mushrooms that are within one or two days of being harvested.

Chemical control — Chlorinated water may lower the bacterial population on the mushroom surface and thereby reduce the amount of blotch; however, chlorine alone is not a cure for this disease. Growers should consult the Health Protection Branch, Health Canada, for guidelines on the use of chlorinated water on mushrooms. The use of chlorine may negate the effectiveness of fungicides previously applied to the casing surface through a process of chemical inactivation. Proper management humidity and recommended watering practices also must be followed.

Selected references

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.2 Mummy *Fig. 26.2*

Pseudomonas sp.

Mummy disease is frequently encountered in mushroom crops; however, an assessment of annual economic loss is not available. When present, the loss is typically only in a portion of any mushroom production chamber.

Symptoms There is no known effect of this disease on spawn or case run, but once fruiting has begun the symptoms appear. The first symptom may be a delayed first break. Mushrooms affected by mummy disease are characterized by curved stems with tilted caps *(26.2).* At the base of the stem, the rhizomorphs are stringy and cling to the casing. The base is frequently swollen and covered with a fluffy growth of mycelium. When harvested, a large amount of casing adheres to the base. Affected mushrooms die, become dry and are tough and leathery. Harvesters can often detect the disease by the tough feel of the stems when cut. Internal stem tissue will often have longitudinal brown streaks or be discolored, and when cut across, minute brown spots can be seen.

Causal agent The pathogen is suspected to be a fluorescent pseudomonad, proximate to *Pseudomonas tolaasii.* Goor *et al.* (see Selected references) provided discriminating characteristics for the mummy pathogen, but the pathogenicity of these cultures from the National Collection of Plant Pathogenic Bacteria, Harpenden, England, was not verified. Other than the original report on causation by Schisler *et al.* (see Selected references), no one has satisfied Koch's postulates with any isolate from symptomatic mushrooms and it has not been possible to induce disease with the reference culture ATCC 25415 from the American Type Culture Collection, Rockville, MD. Disease symptoms are distinctive and reliable for diagnosis. However, confirming the identity of the pathogen requires further study.

Disease cycle The pathogen spreads intracellularly through infected mycelium and not by spores. In a shelf system, the rate of spread is quite rapid, 10 to 30 cm per day. In the tray and bag systems, where mushrooms are cultivated in more discrete units, this symptom is less apparent. However, once the disease develops, the area will not produce harvestable mushrooms. Mummy is often observed in crops where the compost is exceptionally wet after pasteurization and especially when casing is applied over compost in which excess water has accumulated during the spawn-run period.

Management

Monitoring — Compost should be examined for wetness after the pasteurization of phase II compost. Wet areas on the compost surface at casing time or areas where the casing dries rapidly should be carefully examined for the disease.

Cultural practices — Once mummy has been identified, diseased areas should be isolated from uninfested ones by completely removing the compost at least 1.5 m on either side of the affected area to a width of 20 cm and covering them with plastic. At the end of the crop, the compost should be thoroughly pasteurized. Netting and shelving should be carefully cleaned and disinfested before re-use.

Chemical control — Infested netting and shelving should be treated with a formalin solution or hydrated lime.

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(Original by D.L. Rinker and P.J. Wuest)

FUNGAL DISEASES

▶ 26.3 Cobweb (soft mildew) *Fig. 26.3*

Cladobotryum dendroides (Bull.:Fr.) W. Gams & Hoozemans
(syn. *Dactylium dendroides* (Bull.:Mérat) Fr.)
(teleomorph *Hypomyces rosellus* (Albertini & Schwein.) Tui.)

Cobweb occurs infrequently. However, it occasionally can be widespread and destructive on individual mushroom farms.

Symptoms Cobweb disease occurs only on the casing material and may appear at any stage from pinhead onward. Circular patches of white mycelium attack the mushrooms and cover them with a coarse, white growth *(26.3).* Affected mushrooms turn brown and rot. Cobweb mycelium turns pink or red as it ages.

Causal agent *Cladobotryum dendroides* grows rapidly on malt, oatmeal, and potato-dextrose agar. Growth is aerial, cottony and grayish, and a red pigment develops in the medium after five to seven days. Conidia (26 to 32 by 10 to 13 µpm) are two- or three-celled, hyaline, and originate on erect, simple or branched conidiophores (annellophores).

Disease cycle The pathogen is a soil-inhabiting fungus and can be introduced into the casing with soil, spores or mycelium. If the casing is contaminated with spores, symptoms will not develop until the fourth or fifth break. However, when the casing is infested with mycelium, symptoms may occur on the first break. Humidity greater than 90%, air temperatures greater than 18°C, and water condensation encourage the growth of cobweb.

The pathogen is spread via air-borne spores, workers and infested casing material. Wild mushrooms can serve as a host and reservoir for the pathogen. Inadequately pasteurized post-crop compost can serve as a medium for pathogen growth and reproduction.

Management (see verticillium disease, 26.8.)

Selected references

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.4 Green mold *Figs. 26.4a,b*

Trichoderma harzianum Rifai (teleomorph *Hypocrea vinosa* Cook)
Trichoderma koningii Oudem.
(teleomorph *Hypocrea ceramica* Ellis & Everh.)
Trichoderma viride Pers.:Fr.

(teleomorph *Hypocrea rufa* (Pers.:Fr.) Fr.)

Green mold is frequently grouped with the non-infectious molds as an indicator of compost quality. However, some species can reduce crop yield and quality.

Symptoms The white, fluffy mycelial growth will turn green as spores are produced *(26.4a,b).* Sporulation may be observed on compost before casing is applied, as well as on the casing material where it can produce symptoms that resemble cobweb mildew. *Trichoderma koningii* tends to sporulate late in the crop towards third break, whereas *T. viride* will sporulate any time during the cropping period. The pathogen may attack the mushroom cap, resulting in a reddish- or purple-brown coloration that can be confused with symptoms of verticillium spot.

Red pepper mites (see Mite pests, 26.32) are a good indicator species of green mold growing in the compost. They feed on the spores and mycelium of *Trichoderma* and large populations can build up in an infected crop. When mushrooms are fruiting, these red-colored mites will usually aggregate on the cap surface.

The use of protein supplements at spawning or casing can stimulate the growth of *Trichoderma,* especially if the distribution of the supplement in the compost is not uniform or is clumped.

Causal agent *Trichoderma* spp. grow very rapidly on general isolation media. The growth is appressed, sometimes tufted and usually covers a petri dish in a few days. The vegetative mycelium is septate, hyaline and initially gray to white. The colony appearance changes to gray-green when conidia develop. Phialides arise alternately, in pairs or in verticils, and are usually at right angles to the parent branch. Phialides are narrow, 25 to 70 by 2 to 5 pm, with a distinctive taper at the apex where the conidia are borne in masses, 10 pm in diameter. Conidia are hyaline or green, non-septate, smooth, small, and either globoid to ovate or oblong to elliptical, depending on the species. The relative importance and pathogenicity of *Trichoderma* spp. associated with this disease require further study.

Disease cycle The optimum temperature for growth of *Trichoderma* spp. is 22 to 26°C. Sporulation can be observed within 10 days of infestation. The fungus grows particularly well in substrates where the pH is below 6. Compost with a carbon to nitrogen ratio (C:N) of 22:1 will favor development of the pathogen. Normal compost at spawning should have a C:N ratio of 15:1 to 18:1.

Trichoderma species are readily found in soil and on organic matter. The spores are easily dispersed by air currents, water, mites and mechanical means. They can infest mushroom crops at any stage of production.

Management

Cultural practices — Careful preparation of compost will reduce green mold in mushroom crops. A carbon: nitrogen ratio of 15:1 to 18:1 should be achieved. Attention to quality control throughout the compost-making process will produce a properly digested, balanced and selective compost. Supplements should be thoroughly mixed to avoid clumping.

Spawning in a clean-air environment reduces the likelihood of infestation by *Trichoderma* spp. Direct infection of mushrooms can be reduced by lowering the humidity in the growing room.

Standard post-crop pasteurization (65°C) for eight hours is also recommended. Production rooms where crops were severely affected by green mold should be re-steamed before re-filling for the next cropping cycle.

Chemical control — Registered fungicides are available in Canada.

Selected references

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.5 Mat and confetti *Fig. 26.5*

Mat (vert de gris)

Chrysosporiwn luteum (Cost.) Carm.

Confetti

Chrysosporium merdarium (Link:Grev.) Carm.
(teleomorph *Gymnoascus uncinatus* Eidam)

Mat and confetti diseases occur infrequently, and losses to the mushroom industry as a whole are not known. However, losses on individual farms may be quite severe. Mat disease also is called vert de gris.

Symptoms Primordia (pins) do not form in the casing and, consequently, mushrooms do not develop. The cottony, white mycelium of C. *luteum* may aggregate in a distinctive matted layer located between the compost and the casing. Confetti-like

mycelial mats interspersed throughout the compost are caused by C. *merdarium.* These small mats yellow as they age *(26.5)* and may be difficult to see.

Causal agent *Chrysosporium luteum* has irregularly branched mycelium. Small oval conidia, 3.0 to 4.5 µm, are borne irregularly on swollen cells, and short chains of two or three conidia may be produced on a pedicel. Cultures are white when young, but yellow after a few weeks incubation.

Conidia of *C. merdarium* are typically formed on lateral branches as alternate arthroconidia; they are subglobose or pyriform with a broadly flattened base or cuboid when intercalary, smooth-walled to conspicuously roughened, and mostly 5 to 6 by 4 to 5 µm. Colonies on potato-yeast-extract agar develop intensely yellow centers that turn green or reddish brown after three to four weeks.

Disease cycle The two species of *Chrysosporium* infest compost at spawning, ramify concurrently with mushroom spawn, and infect after the casing is applied. Both species tend to be more serious when mineral soil is used as casing rather than peat moss. Formation of mats, within the compost for confetti disease, and on the surface of the compost for mat disease, reflects the growth stage of the pathogen and its interaction with the mushroom mycelium. Little evidence exists on the nature of pathogenesis, but it is thought that secondary metabolites interfere with the formation of the mushroom primordia.

The spores of these pathogens are very small and readily air-borne. Initial infestation of compost likely occurs from sources outside of mushroom operations, for example, when surrounding fields are plowed or eroded by wind. Infested compost can also be a source of large amounts of inoculum. Compost exposed to inoculum of *Chrysosporium* when it is spawned will be thoroughly infested and crop losses will result.

Management

Cultural practices — Mat and confetti diseases, when present in compost, can only be managed through pasteurization (12 hours at 70°C) before the compost is removed from the growing rooms. *Chrysosporium* species are more difficult to control than other molds, hence the need to use a higher-than-normal temperature and a longer time for pasteurization. Otherwise, disease management is based on reducing the number of spores released into the air in the vicinity of the spawning operation. High efficiency particle (HEPA) filters and positive air pressure should be used in the spawning area to minimize infestation of the compost.

Selected references

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.6 Sepedonium yellow mold *Fig. 26.6*

Sepedonium niveum Massee & Salmon (teleomorph *Hypomyces* sp.)

Heavy infestations of yellow mold have been associated with yield reductions.

Symptoms The white mold produced by *Sepedonium* turns dull yellow to tan with age *(26.6).* It competes with the mushroom mycelium. In bulk pasteurization and conditioning methods, it has been found at the bottom layers of the tunnels.

Causal agent *Sepedonium* conidia (aleuriospores) are large (13 to 17 µm), globose, thick- and rough-walled, light yellow and borne singly at the apex of short conidiophore branches. In culture, the thallus is white when young, then turns golden yellow both in and on the substrate.

Disease cycle The spores of *Sepedonium* are resistant to high temperature and may easily survive peak heat. They can spread to the compost by air currents during the filling and spawning operations, or during spawn-run. Unpasteurized or spent compost sticking to beds or trays can spread this mold to the crop.

Management

Cultural practices — Yellow mold can be prevented through careful attention to hygiene and by proper air filtration. Careful monitoring of phase II and postcrop pasteurization temperatures are also necessary.

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.7 Truffle *Fig. 26.7*

Diehliomyces microsporus (Diehl & Lambert) Gilkey

Truffle disease is encountered infrequently in commercial mushroom operations. However, when present, it can cause significant yield losses.

Symptoms Affected areas are circular, about 0.8 to 2.0 m in diameter, and generally exhibit poor growth. Mushrooms on the periphery of such areas experience premature opening of their veils and the stems are thicker than normal; mushrooms may not develop until later breaks. Compost beneath the affected areas becomes soggy, sunken and brown, and the spawn sometimes disappears. Ascocarps (truffles) of the causal fungus develop in infested compost as early as the time of spawn colonization or as late as third break, depending on spawn strain.

Causal agent *Diehliomyces microsporus* has creamy-white mycelium (26.7), in which convoluted ascocarps resembling a calf's brain develop. Oval asci contain eight golden brown, smooth-walled ascospores, and each ascospore contains an oil droplet. Ascocarps rarely form in pure culture and usually require the presence of *Agaricus bisporus* spawn to develop.

Disease cycle The pathogen enters production houses either in mud in hay, straw bales or on unpaved composting wharves, or as air-borne inoculum from previously infested compost. This fungus grows concurrently with the vegetative mycelium of *Agaricus bisporus,* forming ascocarps throughout the compost when its thallus reaches a certain level of maturity. Ascocarps will not form in the presence of all mushroom strains, which can make it difficult to confirm its presence in the compost. It is assumed that this pathogen produces metabolites that inhibit the continued development of primordia into mushrooms.

Not all soils harbor *D. microsporus,* but it is impossible to discriminate between soils that are and are not infested. Once the disease has become established, the pathogen is able to infest wooden shelves or trays and survive in the wood.

Management

Cultural practices — Baled straw should not be allowed to become muddy, and compost should be made on a concrete surface to reduce the likelihood of introducing the pathogen. Wood can be disinfested only by subjecting it to a thorough pasteurization with steam; a temperature of 75°C for 6 to 12 hours is recommended for most woodwork. Infested compost should be pasteurized at 60 to 65°C for 18 hours before it is removed from a growing room. When the pathogen is not killed, propagules can readily contaminate compost during the filling or spawning stages, and infestation at either of these times poses a serious threat to the crop.

Selected references

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Sinden, J.W. 1971. Ecological control of pathogens and weed-molds in mushroom culture. *Annu. Rev. Phytopathol.* 9:411-432.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.8 Verticillium disease (dry bubble, split stipe, verticillium spot) *Fig. 26.8*

Verticillium fungicola (G. Preuss) Hassebring
(syn. *Verticillium malthousei* Ware)

Verticillium disease is the most significant fungal disease of commercial mushrooms, resulting in losses of approximately $7 million annually in Canada.

Symptoms The various names for the disease are descriptive of symptoms resulting from infection at different stages of mushroom development. Early infection of the developing mushroom primordium disrupts its growth, causing it to form a ball-like mass (dry bubble), 0.5 to 1.0 cm in diameter. Infection of older mushrooms (button stage) causes the stem to shatter and the cap may tilt slightly (split stipe). If the pathogen infects the cap tissue, the area turns brown *(26.8)* and the diseased tissue may have a grayish hue (verticillium spot). Lesions are generally less shiny than those caused by bacterial blotch. These brown spots will eventually produce a grayish-white "bloom" of *Verticillium* spores.

Causal agent A selective medium has recently been developed for isolated *V. fungicola* from casing soil, debris in mushroom production rooms, on- farm premises, flies and mushroom sporophores (see Selected references, Rinker *et al.* 1993). It has been successfully used to monitor the efficacy of sanitation programs in commercial mushroom operations in Ontario. *Verticillium fungicola* grows 23 to 28 mm in 10 days on potato-dextrose agar at room temperature and is velvety white in appearance. Undersides of cultures are colorless to yellow. Some isolates grow into the medium and the surface morphology becomes crenate in about 10 days. Mycelium is septate, hyaline, branched and narrow (1 to 3 µm). Numerous phialides are borne in whorls on erect conidiophores, 8 to 10 by 1 to 3 µm, tapering to 0.5 to 1.0 µm at the apex where conidia develop. Conidia form acropetally, but push into a gelatinous matrix rather than being catenate. Conidial masses contain 6 to 20 ovoid to short cylindric to slightly crescent shaped, hyaline, non-sep- tate, smooth-walled conidia, 6.6 by 2.5 µm. Another type of conidium (large aleuriospore) may form in the occasional culture incubated for many weeks. No sclerotia are formed by this species.

Two varieties of *V. fungicola* have been described, but the legitimacy of this differentiation is conjectural. *Verticillium psalliotae* Treschow, another nonsclerotial *Verticillium* species associated with mushrooms, is isolated only from crops cased with topsoil rather than peat moss, when the soil is not adequately pasteurized. A red pigment forms in the medium and the conidia are crescent shaped, but other physical characteristics are very similar to *V. fungicola.*

Disease cycle The optimum temperature for disease development is about 20°C. At this temperature, dry bubble and split stem symptoms will develop in 10 to 14 days from infection. Thus, if bubbles are present at first break, the pathogen was likely introduced with the casing material or at anytime through pin-set initiation. Verticillium spot develops within 48 hours of inoculation. Contaminated casing material and dust are probably the most common sources of *Verticillium.* Contamination may occur through air-borne spores or by spores carried by insects, mites or farm workers. The spores of *Verticillium* are produced in sticky clusters, which enables them to attach to dust, flies, mites, debris, clothing, tools and workers. The sticky spores cannot be removed by washing hands with hot, soapy water. Distribution on the hands and clothing of workers can be the most significant means of transporting the pathogen within a crop or between crops. Watering the crop can distribute the spores through splash and runoff to lower shelves and the floor. Insects, especially phorid flies (see 26.31), and mites (*Tyrophagus* spp.) that feed on *Verticillium* spores can easily move the pathogen within and between crops. Disturbance of contaminated dust on floors increases the concentration of airborne spores and is thought to be the cause of primary outbreaks.

Management

Cultural practices — The strategy for controlling verticillium disease is principally one of good sanitation and hygiene. The basic principles are 1) start with clean facilities and growing media; 2) prevent infection of healthy crops; 3) prevent disease spread within and between crops; 4) keep disease levels as low as possible; and 5) pasteurize to eliminate the pathogen in diseased crops when picking stops.

Casing material should be stored in dust-free areas, and storage and mixing areas should be disinfested before casing preparation. Casing material should not be exposed to dust or flies during mixing. Workers involved in casing application should not have worked in infested rooms or be wearing contaminated clothing. All casing equipment should be sanitized before use.

Insects should be prevented from entering production rooms by physical and chemical means. Controlling fly populations during the cropping period significantly reduces disease spread (see dark-winged fungus gnat, 26.29). Spore filters should be installed on all supply and vent air ducts.

To minimize spread of the *Verticillium* fungus from old (fourth break) to new (first break) rooms, new rooms should be harvested first each day. Harvesters should avoid picking diseased mushrooms. If diseased mushrooms are to be removed, this should be done separately from the regular harvest and workers should wear gloves that are disinfested periodically.

If the disease is severe, the crop should be terminated before the fourth break. The use of a three-break crop can significantly reduce the incidence of dry bubble disease on commercial farms. Crops that have been terminated, either early or on schedule, should have the compost temperature raised to 70°C for 12 hours. Spent compost and debris should be removed from the farm premises. Walls, floors and tray or shelf surfaces should be washed thoroughly after compost is removed.

Chemical control — Registered fungicides are available in Canada; however, benzimidazole-resistant *Verticillium* strains have been reported. Dithiocarbamate products can only be applied as dusts to the casing. Formalin can be mixed into casing material before it is placed onto the spawn-run compost. Local infections can be controlled by covering with salt and/or by cups, or by spraying the infected bubble and surrounding area with formalin before removal.

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(Original by D.L. Rinker and P.J. Wuest)

▶ 26.9 Wet bubble *Fig. 26.9*

Mycogone perniciosa (Magnus) Delacr.
(teleomorph *Hypomyces* sp.)

Although this disease has been found from time to time, it is not a serious problem on commercial mushrooms.

Symptoms The disease is best recognized by the large cauliflower-like distortion of the mushroom *(26.9).* This coral-like mass can measure up to 10 cm across. Under conditions of high humidity, amber to dark brown drops of liquid form on the fluffy white surface. Dry bubbles, caused by *Verticillium fungicola,* do not get as large as wet bubbles and do not turn brown. Under dry conditions, wet bubbles can desiccate and may resemble dry bubbles.

Causal agent Two different types of conidia routinely develop, a phialo- conidium and an aleurioconidium. The phialoconidia are hyaline and are borne on verticillately branched conidiophores. Early research with the phialoconidial stage of *Mycogone perniciosa* resulted in confusion because of its similarity in appearance to *Verticillium fungicola. Mycogone perniciosa* is easily

identified by the presence of aleurioconidia produced on short branches that develop at the base of conidiophores and, more routinely, are intercalary along nonspecialized, narrow (3 to 4 µm) hyphae. Aleurioconidia are borne on a thin-walled, bulbous basal cell, 10 to 14 by 9 to 12 µm. The numerous aleurioconidia, 18 to 20 by 14 by 17 µm, are light amber brown, multinucleate, thick-walled with some warts, and the cytoplasm is dense and granular. Optimum growth occurs at 23 to 25°C on most general-purpose media. There is no light requirement for sporulation and aleurioconidia develop in 7 to 10 days. Phialoconidia are associated with young (3- to 7-day) cultures. Mycelial growth is white. A transformation occurs from white to buff as the culture ages. Aleurioconidia are numerous, while phialoconidia are rarely seen in older cultures.

Disease cycle Contaminated casing material is the primary source of *Mycogone perniciosa*, which is a ubiquitous soil-borne fungus. In young infected mushrooms, the pathogen will take 10 to 14 days to form its distinctive mass. Diseased first-break mushrooms may indicate contaminated casing. Spores may survive on structural surfaces and in crop residue. Once established, the primary means of spread is through water splash and runoff to lower beds. Insects and mites are suspected to be carriers of the pathogen, but there is no firm evidence of this. Harvesters, tools and equipment can spread the pathogen. The spores are light and may be air-borne. Spores in dust on floors or in soil may be another source of contamination.

Management (see verticillium disease, 26.8.)

Selected references

Smith, F.E.V. 1924. Three diseases of cultivated mushrooms. *Trans. Br. Mycol. Soc.* 10:81-97.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.10 Other fungal diseases

Aphanocladium cap spot *Aphanocladium album* (G. Preuss) W. Gams
Gill mildew *Cephalosporium* spp.
Hormiactis cap spot *Hormiactis alba* G. Preuss
Shaggy stipe *Mortierella bainieri* Cost.

These less common fungal diseases occasionally have been reported to cause significant yield loss to commercial mushroom production.

Selected references

Flegg, P.B., D.M. Spencer and D.A. Wood, eds. 1985. *The Biology and Technology of the Cultivated Mushroom.* J. Wiley & Sons, Chichester, England. 347 pp.

Fletcher, J.T., P.F. White and R.FI. Gaze. 1989. *Mushrooms: Pest and Disease Control.* 2nd ed. Intercept Ltd., Andover, Plants., England. 174 pp.

(Original by D.L. Rinker and P.J. Wuest)

VIRAL DISEASES

▶ 26.11 Miscellaneous viral diseases *Fig. 26.11*

La France
Other viral diseases

To date, five or more types of virus particles have been reported to infect commercial mushrooms. Singly and in combination, they produce a variety of symptoms. "Virus disease" frequently occurred in the commercial mushroom industry during the 1960s before its etiology was understood. La France continues to be a serious threat. In many cases, viral diseases are so devastating that mushroom growers have to cease production temporarily in order to eradicate the problem.

Symptoms The symptoms of viral diseases vary from reduced yields to distorted mushrooms. During the spawn- run period, there is no visible indication of disease; however, once the casing has been applied, distinctive symptoms may be expressed. The mycelium may have difficulty growing into the casing in some areas or will grow and then die back, leaving patches with no mushrooms *(26.11).* Mushrooms that do form may 1) be normal, 2) have small caps on normal-sized stems, 3) have elongated stems that are slightly bent, 4) die rapidly followed by a bacterial soft rot, 5) open prematurely, 6) turn off-white, ashen, or tan in color, 7) pin later than normal and frequently below the surface, 8) turn brown rapidly when harvested, or 9) be loosely attached to the casing. In other cases, the crop may appear completely normal, the only effect being an unexplained drop in production.

Causal agents There is a strong correlation between the presence of a specific double-stranded RNA pattern and the symptoms of La France disease. This pattern consists of dsRNA with molecular weights of 2.50, 2.05, 1.95, 1.85, 1.70, 1.10, 0.89, 0.53, and 0.50 x 10^6. Molecular weights of the dsRNAs are estimated from their electrophoretic mobilities relative to the Bst E II restriction endonuclease fragments of lambda DNA. Healthy mushrooms, including the newer hybrid strains, contain another dsRNA, molecular weight 1.6 x 10^6, that is not associated with the presence of disease.

Mushroom viruses (MV) also have been classified or recognized according to the size and shape of the particles. According to Fletcher *et al.* (1989; see Additional references), these include MV1, spherical particles, diameter 25 nm; MV2, spherical

particles, diameter 29 nm; MV3, bacilliform particles, 50 by 19 nm; MV4, spherical particles, diameter 35 nm; MV5, spherical particles, diameter 50 nm.

Positive identification of mushroom viruses is available through comparative growth rates of mycelium on agar, direct electron microscopy (EM), immunosorbent electron microscopy (IEM or ISEM), polyacrylamide gel electrophoresis (PAGE), and enzyme-linked immunosorbent assay (ELISA).

Disease cycle Viral diseases are transmitted through hyphal fusion (anastomosis) of healthy and diseased mycelium; the latter may produce mushrooms that release virus- infected spores. There are no known vectors (insects, mites or nematodes) of mushroom viruses other than the indirect distribution of spores by insects or mites. Mushroom spores can easily become air-borne and thus carried about the room or farm. Wild mushrooms have not been shown to be hosts of the viruses that attack *Agaricus bisporus.*

The compost can be infested any time after peak heat. Once established in the crop, the virus can spread through the mycelium. One mushroom with an 8 cm cap can discharge approximately 1.3 billion spores. It has been reported that as few as 10 to 100 virus-infected spores over approximately 3 m^2 of compost surface will induce recognizable symptoms.

Virus-infected mushroom spores in a dry state and stored at room temperature are capable of transferring virus to healthy mycelium after six years. Mushroom spores stored at 4°C are still viable after 10 years. Research has shown that mushroom spores survive 16 hours at 60°C but not at 65°C. Other reports suggest that 54°C for 10 minutes is lethal to mushroom spores.

Mycelial fragments greater than several cells remaining on woodwork, netting, or equipment can anastomose with healthy mycelium and transfer the virus particles.

Management

Cultural practices — The successful control of mushroom viral diseases can be accomplished through a strict hygiene program (see verticillium disease, 26.8). Since spores are a major vector of viruses, diseased mushrooms must not be allowed to open and release their spores. Filters must be fine enough to capture the 5 by 7 pm spores. Ventilation systems must be tight and not create a negative pressure, thereby sucking spores in beyond the filter.

The technique of ruffling the casing, which consists of breaking up the mycelium and relocating it within the casing layer, emphasizes the need for disinfesting equipment between each use. Also, the new technique of adding fully colonized compost to the casing material ("cacing") increases the risk of disease spread and highlights the need for a comprehensive program of preventive hygiene on mushroom farms.

Changing spawn strains can be helpful in restoring yield. Selecting a strain that does not anastomose as readily with the infected strain can help reduce the inoculum on the farm. Thus, common commercial practice has been to grow a strain of a different color or texture, such as cream or off-white. A related species, *A. bitorquis,* is reported to be tolerant to viral diseases. If hybrid strains of *A. bisporus* are used, switching from white to off-white hybrids or the reverse may not be effective because the hybrid strains are reported to anastomose with their parent lines.

Storage and shipment of spawn and mushrooms in the same cooler should be avoided. If equipment or picking/shipping containers are shared between farms, they should be disinfested before use.

Selected references

Morris, T.J., and J.A. Dodds. 1979. Isolation and analysis of double- stranded RNA from virus-infected plant and fungal tissue. *Phytopathology* 69:854-858.

Romaine, C.P., and B. Schlagnhaufer. 1989. Evidence of double stranded RNAs in healthy and La France disease-affected basidiocarps of *Agaricus bisporus. Mycologia* 81:822-825.

Romaine, C.P., P. Ulhrich and B. Schlagnhaufer. 1993. Transmission of La France isometric virus during basidiosporogenesis in *Agaricus bisposus. Mycologia* 85:175-179.

Ross, R.C., G.A. Brown and C.P. Romaine. 1987. Recent experience in detecting viral double-stranded RNA in commercial mushroom crops and its effect on yield. *Dev. Crop Sei.* 10:321-329.

(Original by D.L. Rinker and P.J. Wuest)

WEED MOLDS

There are several non-infectious fungal diseases of commercial mushrooms, all of which are usually referred to as indicator or weed molds. Production can be significantly affected by their presence, but the mushroom itself is not generally infected.

A. MOLDS CHIEFLY IN COMPOST

▶ 26.12 Ink caps *Fig. 26.12*

Coprinus comatus (Müller in Fl. Dan.:Fr.) S.F. Gray
Coprinus niveus (Pers.:Fr.) Fr.

Symptoms The mycelia of these *Coprinus* spp. are fine, gray to white, and not easily distinguished from mycelium of the mushroom fungus. Fruiting bodies of *Coprinus* spp. *(26.12)* generally develop after casing and before mushrooms are produced. Occasionally, they are observed at the end of phase II composting. The fruiting body quickly degenerates into a black, inky slime.

Causal agent *Coprinus* spp. are periodically seen in mushroom compost, first appearing during spawn-run or after the casing (peat moss) has been applied (top-dressed) to the compost after it has been colonized by *Agaricus bisporus.* These black-spored mushrooms usually have conical pilei and are most easily recognized when deliquescence transforms the mushrooms into a black liquid, which is the basis for the common name "ink caps." Morphological traits, besides deliquescence, that distinguish *Coprinus* spp. from other mushrooms include widely spaced narrow gills, pileus tissue quite thin, very thin lamellae with parallel sides, basidia separated by paraphyses that are shorter and broader than the basidia, and smooth basidiospores with a pore at the apex. At least two species, *Coprinus niveus* and C. *comatus*, have been observed on North American *Agaricus* mushroom farms.

Coprinus comatus originates within the compost and pushes its way to the surface, either before or after casing is applied. Its distinctive, musty, somewhat sweet odor permeates the growing room before it is seen, and the unique odor of the deliquesced mushrooms remains for a few days after deliquescence has occurred. The pileus is 2 to 8 cm high, the stipe is 8 to 20 cm long by 10 to 15 mm thick, and the elliptical spores are 13 to 16 by 7 to 8 µm. The caps are narrowly conical, expand to a bell shape, and are covered with scales that turn brown-red with age and become recurvate.

Coprinus niveus grows on the compost surface in small lumps of black compost, or on the surface of the casing at about the time *Agaricus* primordia are developing. Its primordia develop in 24 to 36 hours and mature after a similar amount of time. Only one bloom occurs, always before the mushroom harvest begins. After deliquescence, no trace remains of C. *niveus.* The caps are 10 to 30 mm high by 10 to 20 mm wide at the base and they are narrowly conical. The stipe is 3 to 6 cm high by 2 to 9 mm thick. Basidiospores are 15 to 17 by 8 to 11 µm and elliptical.

Ink caps bloom within a few days of each other, always before the mushrooms, and should be removed to eliminate the odor and the spore load and to prevent discoloring the mushrooms.

Disease cycle The presence of ink cap fungi indicates insufficiently converted nitrogen-containing compounds in the compost. This may result from an imbalance of the carbon: nitrogen ratio (C:N), overly composted material, too wet or too compacted compost at filling, too dry compost, or in an increase in the compost temperature of as little as 1 to 2°C during phase II composting. *Coprinus* spp. can readily use free ammonia and have an optimum pH for growth near 8. Greater than 700 ppm of ammonia at spawning can stimulate ink cap production. Once the free ammonia is released and the pH declines, mushroom mycelium will colonize the area. Spore masses released by mature ink caps can infect freshly prepared compost.

The presence of a few ink cap fungi has no effect on mushroom yield. It indicates that the compost contained a desirable nitrogen content, but that the ammonia had not completely dissipated before spawning.

Management

Cultural practices — Ink cap fungi can be controlled through proper composting. Attention to compost formula and the composting process is critical to achieving a well-balanced C:N ratio, optimum moisture level, and proper breakdown. Uniformity in the filling process and control over the environment during phase II composting will minimize ink cap infestation. The odor of ammonia should not be present in compost that is ready for spawning.

Selected references

Miller, O.K., Jr. 1984. *Mushrooms of North America.* E.O. Dutton, Elsevier-Dutton Inc., New York. 368 pp.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.13 Olive-green mold *Fig. 26.13*

Chaetomium globosum Kunze:Fr.
Chaetomium olivaceum Cooke & Ellis

Symptoms The mycelium in the compost is grayish- white and fine. If plastic is not used on the compost surface after spawning, a fine aerial growth on the surface may be visible after 10 days; it has a distinctive moldy odor. Within 14 days of spawning, olive-green burs (perithecia) visible to the naked eye will appear on the straws *(26.13).* The mold will frequently occur in black areas of the compost that are uncolonized by the mushroom fungus.

Causal agent *Chaetomium globosum* is homothallic. Its perithecia are dark brown to black (olive-green on mushroom compost) and 225 to 350 µm. The asci are clavate and contain dark, olive-brown spores when mature. The ascospores are lemon-shaped, and flattened, with dimensions 8.5 to 11.5 by 7.0 to 8.5 by 6.5 to 7.5 µm. *Chaetomium olivaceum* has larger ascospores, 8.7 to 12.0 by 8.7 to 10.0 µm; it also is homothallic.

Disease cycle Spores of the olive-green mold fungus are quite common and can be found in straw, soil and spent compost. The ascospores can be carried along in air currents and on clothes and materials. They are heat tolerant and can survive 60°C for six hours. The development of olive-green mold is favored by a lack of oxygen (less than 16% oxygen) during phase II of the composting process. This may occur when the compost is too wet, too compacted at filling, overcomposted, or over-heated

during peak-heat (temperatures greater than 62°C with insufficient aeration). Olive-green mold is able to tolerate higher levels of ammonia than the mushroom mycelium. Thus, it can survive and thrive in conditions adverse for spawn growth.

Mushroom yield may be affected proportionally to the amount of compost that is affected, although different mushroom strains respond distinctively to olive-green mold.

Management

Cultural practices — Once the compost is infested, it is not possible to control olive-green mold. Reheating or re-spawning the compost is not effective. The best means to manage the problem is to avoid conditions that favor its establishment. Preparation of good compost during phase I, attention to filling trays, shelves or tunnels, and providing adequate aeration during phase II composting will prevent the occurrence of olive-green mold.

(Original by L. Rinker and P.J. Wuest)

▶ 26.14 Penicillium mold

Penicillium janczewskii Zaleski
(syn. *Penicillium nigricans* Bainier in Thom.)

Symptoms *Penicillium janczewskii* is another of the green molds that can occur in the compost and on the casing soil. Colonies are usually green but also can be blue- green, white, yellow or brown. *Penicillium* species are opportunistic fungi. They prefer simple carbohydrates but will grow on cellulose, fats and lignin. Poorly mixed or clumps of supplements, dead pins and cut stumps are common sites for the growth of this mold.

Causal agent *Penicillium janczewskii* is frequently identified from collections at mushroom farms. *Penicillium* species appear innocuous to crop health and mushroom quality, but no one has investigated either aspect in detail. The mycelium grows well on general fungal growth media. Colonies reach about 3.0 cm in diameter in 12 days at 25°C. The colonies are feltlike, light to dark olive-gray above and yellow to orange beneath. The conidia are globose, coarsely warted and measure 3.0 to 3.5 µm in diameter. When observed microscopically, the phialides have a tapering apex and conidia are brown pigmented. Several *Penicillium* spp. are associated with mushrooms, wooden boxes and shelves used to hold compost, and with nutrient supplements added to mushroom compost.

Disease cycle Compost can be infested by air-borne spores at spawning and colonization occurs thereafter. From time to time, *Penicillium* spores may contaminate the grain substrate used in spawn, but this is considered to be a secondary source of infestation.

Management

Cultural practices — Follow the same sanitation and hygiene methods used to manage green mold (*Trichoderma* spp.), La France and mat diseases.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.15 Other molds in compost

During composting, many mesophilic and thermophilic actinomycètes and fungi are part of the conversion process. The actinomycetes include species of *Streptomyces, Thermoactinomyces* and *Thermomonospora.* The fungi include *Humicola* (or *Thermomyces*) spp., *Stilbella thermophila,* and species of *Mucor, Thermoascus, Torula, Myriococcum, Malbranchea* and *Talaromyces.* The visible presence of these microorganisms at the time of spawning is a positive indication of the composting process.

(Original by D.L. Rinker and P.J. Wuest)

B. MOLDS ON COMPOST AND CASING

▶ 26.16 Black whisker mold *Fig. 26.16*

Doratomyces microsporus (Sacc.) F.J. Morton & G. Smith

Symptoms This mold can easily be recognized by gray- black, spore-bearing bristles (2 mm) on the surface of straws or casing material *(26.16).* Heavily infested compost will appear gray to black because of the high density of spores. When disturbed, the spores are released, resembling smoke. *Aspergillus, Penicillium* and *Chaetomium* mold species also may be present.

Causal agent *Doratomyces microsporus* conidiophores (annellophores) are aggregated into ereet, black synnemata, up to 600 pm, each having a feathery spore-bearing head. Each head consists of anastomosing hyphae that branch toward the outside and bear numerous conidiophores. Conidiophores are short, 4 to 9 by 3 to 4 pm, and produce long chains of conidia. Conidia are globose to ovoid, smooth-walled, and 3 to 5 by 2 to 3 pm.

Disease cycle Whisker mold is a cellulolytic fungus. The mold will develop when compost has been under-composted, where the carbon to nitrogen ratio (C:N) at spawning is above 18:1, or where the compost has overheated during the spawn-run period. Secondary infection by whisker mold in a crop is unlikely. Human allergic responses to the spores have been reported.

Management

Cultural practices — Attention to proper compost preparation during both phase I and phase II will prevent the occurrence of black whisker mold.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.17 Brown mold

Oedocephalum glomerulosum (BulLChev.) Sacc.
(teleomorph *lodophanus testaceus* (Moug.:Fr.) Korf)

Symptoms Brown mold is silvery gray initially, but as the spores mature the color changes to a dark tan, beige, or light brown. The growth on straws may be sparse to dense. It grows slowly through the casing, appearing near the time of pinning. The spores of this mold feel gritty compared to the smooth, flour-like feel of a plaster mold.

Causal agent *Oedocephalum glomerulosum* has determinate conidiophores that are erect, simple, hyaline, septate and enlarged at the apex into ampullae. Globose, non-septate conidia are produced synchronously on denticles over the surface of the terminal ampullae.

Disease cycle The spores are common in some composts. Development of this fungus is encouraged if ammonia and amines are not eliminated during phase II composting.

Management

Cultural practices — Techniques favoring the production of good compost will reduce the occurrence of this fungus.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.18 Lipstick mold *Fig. 26.18*

Sporendonema purpurascens (Bon.) Mason & Hughes (syn. *Geotrichum candidum* of authors, not Link)

Symptoms Lipstick mold appears on the compost during spawn-run or on the casing during production. This white fungus is not easily distinguished from mushroom mycelium. Cottony white balls, resembling mushroom pins, may develop on the straw or casing surface. As the spores mature, however, a distinctive pink to cherry-red color develops *(26.18).* In the Netherlands, where serious viral infections have occurred, this fungus has also been observed as a secondary mold. In Australia, it has been suspected as a vector of the La France pathogen.

Causal agent Some authors have regarded *Geotrichum candidum* and *Sporendonema purpurascens* as synonyms. However, according to van Greuning and Eicker (see Selected references), the two genera are distinct and the correct identification for lipstick mold is *S. purpurascens.* Lipstick mold can develop on both casing and compost, producing its distinctive red conidia (arthroconidia). Conidiophores are lacking. Vegetative hyphae are hyaline when young but turn brown with age. Conidia are produced by basipetal septation and fragmentation of vegetative hyphae. Small vestiges of cell walls remain on the four corners of the uniformly sized conidia. Germination of conidia has never been observed in culture. This fungus is very difficult to culture and grows very slowly. Growth is diffuse and an irregular, red-black pigment is formed in agar. Another type of conidium (aleuriospore) occasionally forms in cultures more than two months of age.

Disease cycle Lipstick mold is often associated with old, decomposed chicken manure in the compost formula and with wet compost. The fungus tends to spread slowly but it can colonize well-conditioned compost. Dissemination is through spores and any means that transmits them can spread this organism. Its appearance during spawn-run may result in lowered yields, but its occurrence during the harvest period does not affect yields.

Management

Cultural practices — Some authorities recommend raising the compost temperature to 65°C during peak heat for four hours to control lipstick mold. However, this practice can increase the chances of other molds developing. The best methods to reduce the risk of this fungus are to monitor the compost quality and to attend carefully to hygiene and phase II composting (pasteurization) temperatures.

Chemical control — Formalin can be used to spot-treat small areas of lipstick mold infestation.

Selected references

Van Greuning, M., and A. Eicker. 1991. The identity of the lipstick mold of cultivated mushrooms, *Agaricus bisporus. Bot. Bull. Acad. Sin.* 32:57-62.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.19 Plaster molds *Fig. 26.19*

Botryotrichum piluliferum Sacc. & March.
(teleomorph *Chaetomium piluliferum* J. Daniels)
Papulaspora byssina Hotson
Scopulariopsis brevicaulis (Sacc.) Bainier
Scopulariopsis fimicola (Cost. & Matr.) Vuill.
Trichothecium roseum (Pers.:Fr.) Link
(teleomorph *Hypomyces trichothecioides* Tubaki)

Symptoms White plaster mold, *Scopulariopsis fimicola*, may appear on the compost surface near the end of phase II composting as irregular patches of white, filamentous, aerial growth. After spawning, the aerial growth disappears and the white mold becomes appressed to the compost surface, appearing like flour or plaster of Paris. The mold may grow up through the casing material. Other fungi with similar gross morphology that may occur on or in mushroom compost are *Botryotrichum piluliferum* and *Trichothecium roseum.* There are color differences as these fungi mature. *Scopulariopsis fimicola* remains white, while *B. piluliforum* takes on a tan-buff appearance and *T. roseum* develops a rose-pink tint.

The brown plaster molds *Papulaspora byssina* and *Scopulariopsis brevicaulis* appear during the spawn-run period as 15- to 40-cm patches of a dense, plaster-like white mold. When mature, the center of the colony turns brown or orange-brown *(26.19).* The fungus grows through the casing and develops the characteristic brown center with white fringe. Both fungi grow well in compost with a pH of 8.0 or more.

Causal agents *Scopulariopsis fimicola* is the most commonly encountered white plaster mold in mushroom compost. Its mycelium is colorless, septate, sparsely branched, and 2 to 5 µm in diameter. Conidiophores occur in groups of four or five, separated from each other by long portions of sterile mycelium. They are 50 to 100 µm, cylindrical, taper to the apex, and have irregular, U-shaped, dichotomous branching. Conidiophores initially bear a terminal conidium (phialospore) 6.5 to 8.0 by 4.5 to 5.3 µm.

Conidia (aleuriospores) of *Botryotrichum piluliforum* are globose, 13 to 15 µm, and are produced on repeatedly racemosely branched, hyaline conidiophores.

Colonies of *Trichothecium roseum* are dusty and rose-colored. For a description of this fungus, see Greenhouse cucumber, leaf rot, 22.13.

Scopulariopsis brevicaulis colonies are initially white, but the center develops brown or golden coloration extending to a white edge. Conidiophores are short annellophores, 9 to 25 by 2.5 to 3.5 µm, with the base swollen to 5 µm. Non-septate conidia (amerospores), 5 to 8 by 5 to 7 µm, are globose to ovoid and frequently truncate at the point of attachment. They usually occur in long chains.

The genus *Papulaspora* is characterized by the presence of papulospores or "bulbils" which are sclerotium-like reproductive units consisting of irregular clusters of cells. The bulbils are pale brown to orange pigmented, 100 to 250 µm across, and arise from lateral branches.

Disease cycle These molds are associated with composts where there has been insufficient conversion of the nitrogen sources during phases I and II. They often appear to be associated with compost ingredients.

Management

Cultural practices — Modification of composting procedures for both phases to improve the quality of mushroom compost will significantly reduce the occurrence of the plaster molds.

(Original by D.L. Rinker and P.J. Wuest)

C. MOLDS CHIEFLY IN AND ON CASING

▶ 26.20 Cinnamon brown mold (peat mold) *Fig. 26.20*

Chromelosporium fulvum (Link:Fr.) McGinty, Korf & Hennebert in Hennebert & Korf
(syn. *Botrytis fulva* Link:Fr.)
(teleomorph *Peziza ostracoderma* Korf)

Symptoms Cinnamon brown mold grows mainly on the casing within the first two weeks. On occasion, it has been observed on the surface of the compost during spawn run. It is frequently seen on the surfaces of wooden shelves and trays. The fungus first appears as fine, white, aerial mycelium. The spores form in a few days, which changes the color to light yellow or golden brown. The thick, white, fluffy mycelial edges remain. It has been reported that a dense growth of this mold will retard first break and may cause a slight reduction in yield.

The fungus is opportunistic, not readily tolerating other organisms. It tends to grow on casing that has been overly pasteurized, where a strong formalin solution has been used, or where a virus has killed the mycelium in the casing. The mold will disappear by first break, and 10 to 14 days later the small, dark brown, disk or cup-shaped apothecia of the teleomorph state will appear *(26.20).*

Causal agent *Chromelosporium fulvum* has erect, septate conidiophores with an unbranched main axis bearing 7 to 12 sporogenous ampullae, the spore-bearing heads. Conidia develop simultaneously on denticles located on the surface of each ampulla. The globose conidia are lightly pigmented and tan-brown and cover each ampulla at maturity.

Disease cycle The mold is easily air-borne, which facilitates the contamination of casing material. Since the fungus is not a strong competitor with the mushroom mycelium, secondary infections in a crop are highly unlikely when the mushroom mycelium is healthy. Serious outbreaks on mushroom farms are usually indicative of poor hygiene or improper cultural practices. Cinnamon brown mold is favored by high humidity and high temperature after casing.

Management

Cultural practices — When pasteurizing casing material, careful monitoring of the temperature is necessary to reduce the possibility of over-heating the material. The casing should be heated to 70 to 75°C and maintained at this temperature for 30 min, with a thermometer placed in the coolest area of the pile. Careful attention to hygiene will reduce the risk of spread.

Chemical control — When treating casing material with formalin, no greater than a 2% solution of commercial formalin (37% formaldehyde) should be used.

Selected references

Hennebert, G.L., and R.P. Korf. 1975. The pest mold, Chromelosporium ollare**,** conidial state of Peziza ostracoderma, and its misapplied names, Botrytis crystallina, Botrytis spectabilis, Ostracoderma epigaeum and Peziza atrovinosa. Mycologia 67:214-240.

Stoller, B.B. 1972. The brown mold, Plicaria fulva, growing in mushroom beds. MG **A** Bull. 277:553-561.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.21 Nematode-trapping fungi

Arthrobotrys spp.

Symptoms When nematode populations are high, a superficial, sparse, white growth of the fungus is frequently observed on the casing surface. The affected area can be greater than 1 m in diameter. Some species of *Arthrobotrys* can produce brown colonies on the casing layer. This fungus traps and feeds on free-living, saprophytic nematodes *(Rhabditis* spp.).

Causal agent *Arthrobotrys* colonies are spreading, thin and hyaline or pink. Conidiophores are erect, arising from the substrate or from fasciculate aerial hyphae, and are simple or branched. They produce apical clusters of two-celled, hyaline conidia successively on broad denticles on sympodial branches. Conidial heads often become intercalary by renewed growth of the conidiophore.

Disease cycle Since the fungus needs nematodes to survive, it will appear only in association with heavy infestations and usually near the end of the mushroom crop.

Management

Cultural practices — Sanitation and other practices that deter the build-up of saprophytic nematodes in casing (see 26.28) will help to prevent the growth of *Arthrobotrys* spp.

(Original by D.L. Rinker and P.J. Wuest)

NON-INFECTIOUS DISORDERS

▶ 26.22 Hardcap (hardgill)

The primary cause of this disorder is believed to be the degeneration of spawn cultures. Occasionally, dramatic changes in the temperature of spawn cultures or compost have also been implicated.

Symptoms Affected mushrooms appear normal when viewed from above. From below, however, they are open and lack a veil. The gills are pink or frequently white. Occasionally, the gills are distorted, resembling those of a polypore. The cap is hard and brittle. The mycelium grows well through the compost and casing. First break is delayed and the time between breaks lengthened. The condition occurs throughout the crop. Mushroom production can be reduced to as little as 20% of a normal harvest.

Management

Cultural practices — Growers should follow practices recommended for the maintenance of spawn cultures and the growth of mushroom crops.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.23 Open veil

In some strains of *Agaricus* spp., watering too close to harvest can cause the mushrooms to open prematurely. This often occurs when the mushrooms have been under a water stress and a generous watering follows. Changes in temperature also can trigger opening of the veil, as can excessive carbon dioxide levels during cropping.

Symptoms The cap opens prematurely and the gills are fully developed and brown pigmented. On occasion, the cap is disproportionally smaller than the stem. Open veil sometimes can be a symptom of a viral disease.

Management

Cultural practices — Generally, open veil can be avoided by maintaining suitable growing conditions and by not putting the crop under stress.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.24 Rose comb

Rose comb is associated with hydrocarbons, phenols and other compounds contaminating the casing or contacting the mushroom surface. Diesel oil, exhaust from engines, and petroleum-based pesticides are thought to be the principal source of these chemicals.

Symptoms Pink, gill-like tissue develops on the surface of the mushroom cap. The mushrooms are grotesque and unsaleable.

Management

Cultural practices — Growers should avoid exposing mushroom crops to the harmful chemicals that have been associated with this disorder. To assess their possible toxicity, paints, caulking compounds and other products that are to be used in the growing rooms should be applied to a board and placed next to developing mushrooms. If no symptomatic mushrooms develop, the material is likely safe to use.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.25 Stroma

Stroma is related to the genetic characteristics of the mushroom strain. Some strains will characteristically produce more stroma than others. At other times, however, stroma may be accentuated through mishandling of the spawn in transit, storage or during preparation. Non-uniform casing moisture, especially wet areas, often is associated with the occurrence of stroma.

Symptoms The mycelium on the compost or casing surface aggregates into discrete, white patches, which later develop into a dense layer that can be peeled from the surface of the substrate. The formation of stroma occurs in advance of pinning.

Management

Cultural practices — Spawn should be carefully handled and stored to minimize the risk of this disorder.

(Original by D.L. Rinker and P.J. Wuest)

▶ 26.26 Other abnormalities

There are a number of other abiotic conditions that result in the formation of abnormal fruiting bodies, such as weepers, hollow cores, shaggy stipe, purple stem and saggy socks. Although these conditions are rare, they often concern growers. (For more information, see Additional references.)

(Original by D.L. Rinker and P.J. Wuest)

NEMATODE PESTS

Parasitic and saprophytic nematodes are rarely a problem in mushroom crops grown in modern facilities. Historically, the overall economic impact of nematode pests on Canadian mushroom production has been minimal; however, significant yield losses have been reported occasionally.

▶ 26.27 Parasitic nematodes

Aphelenchoides spp.
Ditylenchus spp.

Extensive sampling of commercial mushroom houses in Canada has not revealed the presence of *Aphelenchoides* and *Ditylenchus* species. For a detailed discussion of these pests, see Hussey *et al.* and Goodey, 26.28.

▶ 26.28 Saprophytic nematodes

Acrobeloides spp.
Caenorhabditis spp.
Choriorhabditis spp.
Rhabditis spp.

Saprophytic nematodes are common in mushroom houses, but there is inconsistency in the scientific literature concerning the correlation of their populations with yield reductions. Nevertheless, most commercial mushroom operations attempt to minimize the incidence of these nematodes.

Damage Black necrotic areas may be visible on the surface of spawned compost prior to casing. These spots may show some colonization by the mushroom fungus, but the mycelium will be fragmented and the compost appears wet. These areas will not be recolonized by mushroom mycelium. The surrounding, colonized compost degenerates and the nematodes subsequently migrate into the casing layer. With careful observation under bright light, the nematodes can be seen with the naked eye as they move or "flicker" on the compost straws.

Often, the casing is well colonized by the mycelium, but after ruffling or scratching it does not re-knit well in spots or in the whole shelf. Sometimes, however, the casing is colonized more slowly than normal after inoculation because of nematode activity. Similar effects have been observed with high populations of entomopathogenic nematodes. In either case, the mycelium is fragmented and the casing does not hold together well. Growers sometimes confuse the dieback symptoms of viral diseases with those caused by nematodes. As in compost, nematode flickering is the key diagnostic feature and may be observed on the casing surface with the aid of a bright light.

The whiteness of the mushrooms may also be reduced. The bacteria on which the saprophytic nematodes feed reproduce well in moist environments. Both the nematodes and their associated bacteria can reduce the quality of the fresh mushrooms.

Identification The majority of saprophytic nematodes in mushroom casing belong to the genera *Acrobeloides, Caenorhabditis, Choriorhabditis* and *Rhabditis.* They are all bacterial feeders and are characterized by having three or six lips fused or replaced by other structures. Their mouth openings (stoma) lack a stylet and their cuticle is annulated or smooth. Amphids are inconspicuous and the oesophagus has a terminal bulb. The tail of the male usually possesses a genital cavity (bursa) supported by rays. There are no caudal glands.

Life history Saprophytic nematodes are common inhabitants of compost and casing mixtures. Under optimum conditions, a 50- to 100-fold increase each week is possible. Under slow drying conditions, especially during the prepasteurization phase of compost preparation, some nematodes can form resistant stages that enable them to survive pasteurization. Insects, equipment, workers and irrigation of the casing can disperse the nematodes. In older mushroom houses with wooden ceilings, nematodes can reproduce in the wet insulation and drop onto the compost in condensation from the ceiling.

Management

Cultural practices — Good sanitation reduces the spread of nematodes in mushroom houses. Thorough, post-harvest pasteurization and cleaning of production rooms, netting and equipment will reduce carryover from one crop to another. Houses with wooden shelving require an especially thorough, post-harvest clean-up because nematodes may be located in the crevices of boards.

Maintenance of proper temperatures during pasteurization of compost is critical. If the surface of the compost becomes dry during the pre-pasteurization phase, resistant stages of the nematodes form and may survive the pasteurization process. Changing the length of the pre-pasteurization period, adjusting ventilation and controlling humidity are also necessary.

Insects are excellent vectors of nematodes. A good integrated pest management program is required to reduce the impact of flies on mushroom crops (see Insect pests, 26.29-26.31).

The casing material can be a source of nematodes. Although peat may be infested with nematodes, packaged peat is generally not a problem if it is handled properly. However, if the bags are broken and the peat becomes wet, the nematodes will multiply. Once opened, the peat should be mixed in a clean area with disinfested equipment and used within 24 hours.

Once nematodes are noticed on compost or casing, steps should be taken to reduce their spread on the farm and to determine the source of the infestation. Tools and equipment should be disinfested between shelves during the spawning operation. Ruffling or scratching the shelves should be avoided where nematodes are visible on the casing.

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(Original by D.L. Rinker and T.H.A. Olthof)

INSECT PESTS

▶ 26.29 Dark-winged fungus gnat *Fig. 26.29T1*

Lycoriella mail (Fitch)

The dark-winged fungus gnat, also known as the sciarid fly, big fly or mushroom fly, occurs across Canada wherever commercial mushroom crops are grown. It is the most important fly pest of commercial mushroom production. Of the three species of mushroom cultivated commercially in Canada, only the button mushroom is subject to severe attack by this gnat. Fly reproduction is continuous because mushrooms are cultivated year-round. In the winter, gnat populations are lowest. Although the threat is minimal in winter, gnats can move from one production room to another through corridors, lofts or outside the building, and they have been seen on the surface of snow 7 to 10 m away from a functional production room. During warmer months, the gnats will move from building to building, and to farms several kilometres distant.

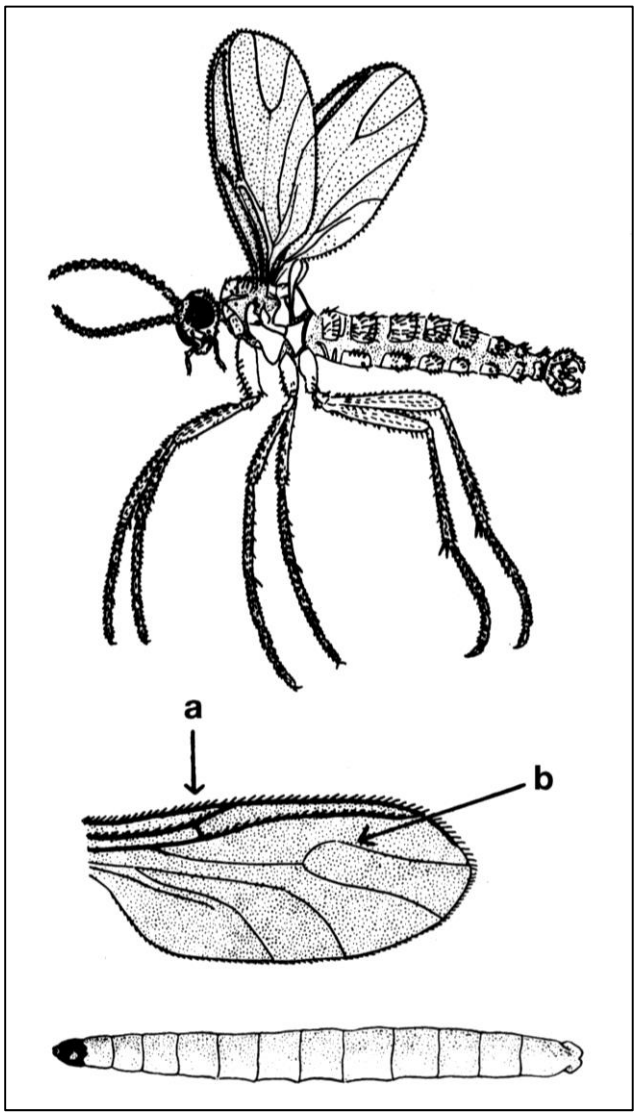
In general, the dark-winged fungus gnat can be found in greenhouses in both soil-free and soil mixtures, in composting debris such as leaves, and outdoors on wild mushrooms. The commercial button mushroom appears to be its preferred host, but it also breeds on oyster and shiitake mushrooms.

Damage The dark-winged fungus gnat can be found on practically any mushroom farm, but direct yield losses occur only when the gnats go unchecked. Larvae of this fly are general feeders, consuming mushroom compost, mycelia, spawn grains, mushroom primordia, and mushroom sporophores. When mushroom primordia are small, up to a cap diameter of about 1.5 cm, the larvae eat the internal contents. The mushrooms will appear glossy and light brown, but the pileus may be completely perforated and, when picked, the tissues crumble. Mushrooms that are larger when attacked show black necrotic areas in the stipe where the larvae made feeding galleries. Some larvae do not tunnel into the stipe but consume the mycelium at the base of the stem, in which case the mushroom does not develop normally. Little direct damage from this gnat will be evident on first-break mushrooms because they are harvested before larval development is extensive. However, second-break mushrooms may show damage from larval feeding. If fly populations are sufficiently high, then larval damage will be visible on the third and subsequent breaks.

The primary disease agent carried by this gnat is that of verticillium disease, which often increases between the second and third breaks. The presence of adult flies during the third break facilitates spread of the disease inside the room. In most cases though, the crop is terminated before a new generation of flies emerges.

This gnat often appears to be an indicator of poor management; as a pest, it is usually only of secondary or minor importance. However, on farms where the gnat is not controlled, individual crop loss can be as much as 50%. Perhaps its greatest impact is as a vector of mushroom pathogens from diseased to clean areas in the same production room or to clean crops in adjacent production rooms.

Identification Eggs of this fly (family Sciaridae) measure 0.25 by 0.15 mm and are smooth, oval, white and translucent. Larvae are legless, about 7 mm long at maturity, and have a white, translucent body and a black head. Pupae (puparia) are 2.0 to 2.5 mm long; they are white at first but turn black prior to hatching. Adult males and females measure 2 to 3 mm in length; most often they can be found near a light source. The wing has a forked vein and a crossvein (*26.29T1*; compare with phorid fly, *26.31T1).*



26.29T1 Dark-winged fungus gnat; adult, 2-3 mm long (top); note crossvein (a) and forked vein (b) in wing (center) and black head capsule of larva (bottom). Adapted from Snetsinger (1972).

Life history This fly usually invades production rooms at or near the time of spawning. After invasion, adults may oviposit on mushrooms, on compost, or in the casing soil, laying eggs singly or in small groups. The larvae have four instars and may feed on the spawn grains, mushroom compost, mycelium and mushrooms. There may be two complete generations during each cropping cycle on commercial mushroom farms; progeny of the second generation tend to be most damaging to the crop. Farms that harvest five flushes commonly have a third generation of fungus gnats.

The optimum temperature for development and maximum survival is 18.3°C. At higher or lower temperatures, mortality increases and female fecundity decreases.

Within four hours of emergence, female gnats are sexually receptive. Egg laying begins soon afterward. Female flies are strongly attracted to odors emanating from the mushroom house, particularly as the compost cools after the pasteurization and conditioning phases. Peak fly-invasion generally occurs within four days of spawning. Flies always seek the nearest site for oviposition; in commercial mushroom operations, this is usually nearest to doors of the production facility. Larvae developing in the surface layer of compost, which is later to be cased, will move through and pupate at or near the surface of the casing layer.

Management The movement of these flies from one production room to another or from one mushroom farm to another is accomplished mainly by unassisted adult dispersal and the persistence of the insects, which will crawl through any crack or crevice into the mushroom-production facility. Under conditions of poor sanitation and hygiene, these flies can enter the room on equipment that was not cleaned from previous spawning or casing procedures. Consequently, by leaving spawn overnight in a fly-infested corridor, an infestation can be introduced into the production room through the spawn. Growers who obtain

mushrooms from other growers run the risk of introducing fungus gnats. The dark-winged fungus gnat is the major insect pest, so management strategies have been developed particularly with this insect in mind.

Monitoring — When an invasion occurs, the size of the initial population and predicting its future size are important considerations. Monitoring gnat populations provides this information, but such factors as climate, disease, immature and adult insect populations, and growing practices are also important. The adult stage of the dark-winged fungus gnat is the main concern when monitoring on a commercial mushroom farm. Monitoring for adults is done by means of a fluorescent or "black" light as an attractant and a sticky surface or pan of water as a trap for the flies. Generally, the wharf end of a room at the upper level has considerably more flies than the breezeway end. In cool weather, the breezeway may serve as a bridge for fly movement between rooms. On standard mushroom farms, the trap should be placed on the wharf wall at the upper level. In the conventional shelf or tray system, the monitor should be placed in the room prior to the compost cool-down period. Monitors positioned outside, where flies normally roost, and in the breezeway, provide an indication of the background or endemic population level and measure the effectiveness of an outside or breezeway control program. Fly catches should be recorded daily to evaluate present control programs and to design future strategies. Threshold levels at different stages of a crop vary. For this reason, growers are encouraged to determine their own economic threshold levels. A suggested action level for the dark-winged fungus gnat is one to two flies each day from before spawning to four days after spawning when the growth room is cooler than 43°C; 15 flies per day for the remainder of the spawn- run; 30 to 40 flies per day after casing; and if a monitor light is used, one fungus gnat at spawning.

Cultural practices — Cropping for only three breaks is a viable, economical management practice because fly and disease problems are reduced. Rapid cool-down at the end of phase II reduces the time available for fly invasion. Higher temperatures occur in the compost during spawn- run and after casing, compared to the harvest period. Shortening the spawn- and case-run prolongs fly emergence in the cycle of the crop's phenology. In general, less damage will occur to a crop if the spawn-run is short.

Fly control at the end of a crop is just as important as control during spawn-run. Growers should treat mushroom houses by steaming-off, or by holding the compost at 60 to 65°C for four hours to kill flies at all stages of development; these conditions also kill most disease-causing fungi and bacteria. A crop may have to be terminated earlier than the schedule dictates to ensure that the population of emerging flies can be controlled prior to its spread to other locations.

Prevention is the most effective way to control fungus gnats. The problem is avoided if adults can be prevented from entering the growth rooms. Cracks in the walls, doors and around air conditioners and pipes are the usual routes of initial fly invasion. The installation of netting over doors, and limiting the amount of traffic into the room at critical times can help reduce the likelihood of infestation. Traps can be used to determine the tightness of a room and the need for doorway management. In general, if flies can be excluded until casing, they will have little or no impact.

Good sanitation is also important for fly control. Flies can breed in the butt and fragments of discarded mushrooms, and spent compost may serve as breeding material. Spent compost and mushroom stumpage should be removed from the premises. Growers also should remove and dispose of trash promptly.

The farm community must also be considered. Each mushroom room, block and farm probably has an endemic or background population of fungus gnats. These populations are specific for each farm and will vary from crop to crop and season to season. Pest populations throughout the farm community can only be brought under control if each grower understands the benefits of a consistent and total fly control program. Cooperation among growers also promotes better gnat management, based on sharing of knowledge about fly biology and behavior, and about the essential conditions that favor colonization and spread of pathogens.

Biological control — *Bacillus thuringiensis* var. *israelensis* and predatory nematodes have provided effective fungus gnat control in research trials. At present, the bacterium is not registered for use on mushrooms in Canada; the use of predatory nematodes does not require registration.

Chemical control — Studies in Pennsylvania and Delaware have demonstrated resistance within the darkwinged fungus gnat population to such insecticides as permethrin and dichlorvos, and there is some suggestion of resistance to the insect growth regulator diflubenzuron. Dichlorvos has been used in Canada for a number of years, and both diflubenzuron and permethrin are in the process of being registered for mushroom fly control in Canada. Many insecticides commonly used in the mushroom industry are metabolized by the enzyme system implicated in permethrin and dichlorvos resistance. Fungus gnats in Canada have not been examined for resistance, but the extensive use of insecticides means that resistance may eventually be present in Canada.

Premise sprays should include resting, swarming and roosting areas during the peak of the fly season. Growers also should treat walls, door jambs and the plastic cover that is placed over the compost after spawning. Adulticides in the form of aerosols or dusts should be applied when the action threshold is reached. For maximum effectiveness, larvicides must be applied when larvae are susceptible; this is especially true for formulations containing the insect growth regulator methoprene, which is a juvenile hormone mimic. For this chemical to be effective, the fourth-instar larva must ingest it; therefore, monitoring populations and maintaining records are essential for accurately timing its application. The use of chemical insecticides can be an important part of fungus gnat control on a farm, but growers should try to integrate this with other practices.

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(Original by D.L. Rinker)

▶ 26.30 Gall midges

Mycophila spp.

Gall midges (cecid flies) are minor pests of mushroom in Canada. The larvae apparently do not become pests on shiitake mushrooms cultivated on a sawdust medium.

Damage Midge larvae feed on the outside of the stipe or at the junction of the stipe and gills of both the commercial button mushroom and the oyster mushroom. Their presence can result in a loss of volume of fresh or processed, marketable product. They are also a factor in the spread of bacteria that induce browning.

Identification Gall midges (family Cecidomyiidae) are small, rarely seen flies, about 1.5 mm in length. However, when populations are high, their larvae are readily noticed because they wander off the beds and accumulate in heaps on the floor. Midge larvae are white or orange, depending on the species; mature larvae are about 2 mm in length.

Life history Gall midges in mushroom cultivation sometimes reproduce by the mature larva giving birth to 12 to 20 daughter larvae without becoming an adult fly and mating. Larvae usually feed for about 14 days, pupate and produce adults in 18 to 21 days. At the initiation of primordia, larvae must feed on mycelium in the casing and on the forming mushrooms. At maturity, the larvae construct pupation chambers in mushroom compost and enter a one-day prepupal stage. After pupation, adults emerge and become active.

Gall midge development is strongly influenced by temperature. During the spawn- and case-run production periods, first-generation flies can emerge within 18 days. During later stages of production, when the substrate temperature is allowed to drop to about 19 to 21°C, the developmental time per generation lengthens to about 21 days.

Management Gall midges are associated with infested casing material, especially peat, and they disperse on inadequately sterilized growing surfaces and especially on tools, equipment, and workers' shoes and clothing. Any practice that minimizes fly dispersal contributes to gall midge control.

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(Original by D.L. Rinker)

▶ 26.31 Phorid flies *Fig. 26.31T1*

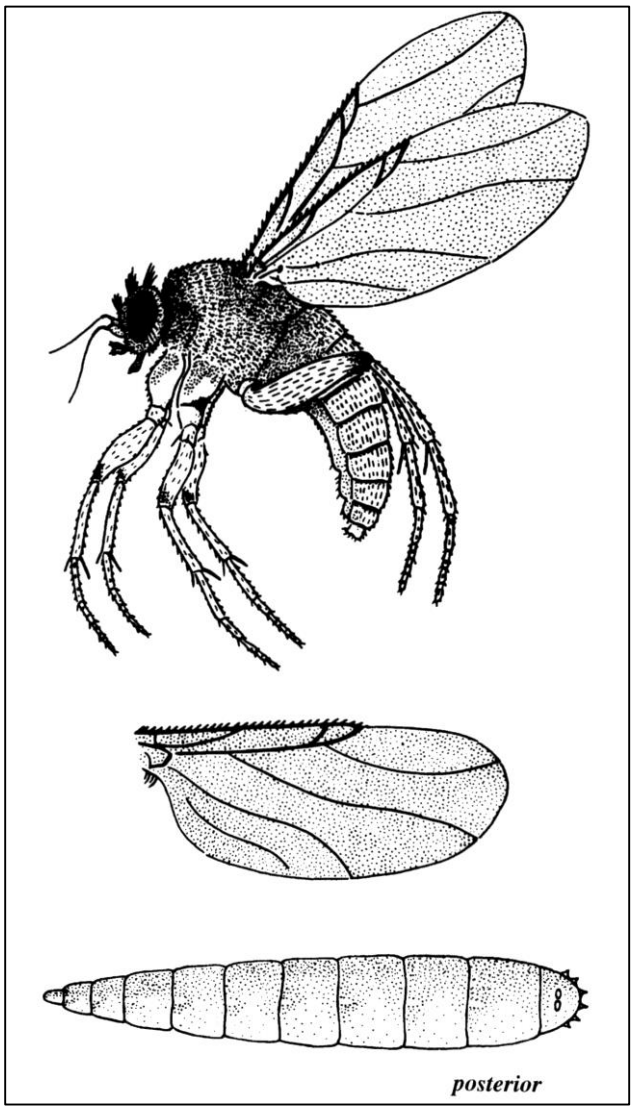
Megaselia halterata (Wood)

Phorid flies have only been observed on one mushroom farm in Ontario. However, they have been a significant problem in the United States and Europe.

Damage *Megaselia halterata* larvae feed at the growing hyphal tips of the mushroom mycelium. This species, unlike other *Megaselia* spp. that were pests in the 1940s in the United States, does not consume the sporophores. Thus, direct yield loss correlates with the number of larvae grazing on the mushroom mycelium. More than 12 000 females per m^2 of production surface are necessary before significant yield loss occurs; this is 12 times greater than the damage threshold for the dark-winged fungus gnat. Although direct yield loss can be a problem, the greater threat is the transmission of *Verticillium fungicola* (see verticillium disease, 26.8).

Identification The eggs of the phorid fly (family Phoridae) are about 0.2 by 0.5 mm in diameter and lack surface sculpture. Fertile eggs are translucent; infertile ones are cloudy and opaque. The legless larvae lack an apparent head and possess posterior respiratory structures (horns), thus differing from sciarid larvae, and the first-, second-, and third-instar larvae have mouthhooks (cephalo-pharyngeal skeletons) measuring 50, 84, 114 µm, respectively. The pupae (puparia) are approximately 2 mm in length. Young pupae are whitish with respiratory horns barely visible. Older pupae are yellow-brown with fully developed respiratory horns. The outline of the adult fly is visible through the puparium near the time of emergence. The adults of both sexes are small, measure 2 to 3 mm, lack forked veins and crossveins in the wings (*26.31T1;* compare with fungus gnat, *26.29T1)*, and are easily recognized by their "humpbacked" appearance, laterally flattened hind femora and quick, jerky movements.

Life history Adult females are attracted to actively growing mycelium and oviposit near the hyphal tips. In commercial operations, the mycelium is actively growing about four days after spawning and after casing. Unspawned compost does not support reproduction. Adult females mate in 24 to 48 hours after emerging (eclosion), and have a two- to three-day pre-ovipositional period before laying about 50 eggs. The average developmental time from egg to adult at 16 and 24°C is 51 and 37 days, respectively, with adults surviving four to eight days.



26.31T1 Phorid fly; adult, 2-3 mm long (top); wings lack cross- and forked veins; note posterior respiratory horns and lack of head capsule on larva (bottom). Adapted from Snetsinger (1972).

Newly emerged and older adult phorid flies readily fly to a suddenly exposed light, especially the shorter wavelengths of black light, black-light blue or cool white. Outside, flight activity is restricted to the daylight hours.

Management The integrated pest management strategies for control of sciarid flies, 26.29, also are effective in managing phorid flies.

Monitoring — Shorter wavelength lights than those used for sciarids are more effective for phorid flies. The action threshold can be at least five times higher than for sciarid flies.

Cultural practices — Since the phorid is smaller than the sciarid fly, the size of screening must be smaller to prevent passage of the fly.

Biological control — The larvae, pupae and adults of *M. halterata* are frequently parasitized by the endoparasitic nematode *Howardula husseyi* Richardson, Hesling & Riding (Tylenchida: Allantonematidae). Parasitism by this nematode does not obviously change the external appearance of the fly or appreciably affect the length of its life cycle; the most significant effect is a marked reduction in fecundity. Laboratory fly populations can be virtually annihilated within five generations by this parasite. Parasitic nematode populations can be favored by preventing compost temperature from exceeding 27°C.

Chemical control — The juvenile hormone mimics and insect growth regulators do not control *M. halterata* populations very well.

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(Original By D.L. Rinker)

MITE PESTS

▶ 26.32 Red pepper mites

Pygmephorus spp.

Red pepper mites, also known as pyemotid or pigmy mites, actually feed on molds (*Trichoderma, Monilia,* and *Humicola* spp.). They seem to be found only in production of the commercial button mushroom.

Damage Red pepper mites do not cause direct damage to cultivated mushrooms, but their presence often contributes to a loss in marketable yield. Additionally, they are a nuisance to mushroom harvesters.

Identification These mites (family Pyemotidae) are tiny, 0.25 mm long, and brown. Because they tend to congregate on top of the mushroom caps, they can be seen by shining a light across the pre-harvest mushrooms.

Life history The mites have a sexual, adult stage and a generation time of four to five days. Adult females may lay up to 160 eggs over a five-day period.

Management These mites disperse on inadequately sterilized production surfaces, on workers' clothing and by clinging (phoresy) on fungus gnat flies. Proper preparation and pasteurization of compost to minimize weed molds will reduce or eliminate red pepper mite populations.

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(Original by D.L. Rinker)

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