8 Crucifers broccoli, Brussels sprouts, cabbage, cauliflower, radish, rutabaga, turnip

Figures *8.1* to *8.47*

Bacterial diseases

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Fungal diseases

8.5 Alternaria diseases
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8.17 Black speck of cauliflower
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Additional references

BACTERIAL DISEASES

▶ 8.1 Bacterial leaf spot (peppery leaf spot) *Figs. 8.1a,b*

Pseudomonas syringae pv. *maculicola* (McCulloch) Young, Dye & Wilkie

Bacterial leaf spot occurs mainly on cauliflower and, to a lesser extent, on broccoli and Brussels sprouts in most crucifer-producing regions. Losses may be significant in isolated fields, but disease outbreaks occur only occasionally.

Symptoms The disease is first seen as spots or lesions on the underside of the outer leaves, which are always more seriously affected than the inner ones. Each spot is associated with a stomate. The lesions are minute at first, about 1 mm in diameter, and brown to purplish *(8.1a)*. Later, a yellow halo develops around each spot as it expands. The spots grow together to form light brown, papery areas that eventually tear, giving the foliage a ragged appearance.

Infection on the veins restricts growth, resulting in puckered foliage. Extensive infection may cause leaves to drop. The pathogen may also cause small, unsightly, gray to brown spots on the curd or head *(8.1b)*, especially during cool, wet weather or after a frost. The affected tissues are firm at first but may become soft if invaded by secondary organisms.

Causal agent *Pseudomonas syringae* pv. *maculicola* is a motile, Gram- negative rod with polar flagella. The cells are 0.7 to 1.2 pm in diameter and about 1.5 pm long. Colonies fluoresce under ultraviolet light on King's B medium. The organism is oxidase and arginine dihydrolase negative and produces levan from sucrose. The strains are nonpectolytic, but they cause a hypersensitive response on tobacco upon infiltration. The pathogen can be isolated from diseased tissue using routine procedures. Pathogenicity must be verified by inoculation onto the host to confirm identification.

Disease cycle *Pseudomonas syringae* pv. *maculicola* survives on infested seed and crop residues. It can remain viable in soil for two to three years. A small percentage of infested seed is sufficient to give rise to an epidemic in the field. The pathogen is spread by splashing water, washing soil and possibly by insects. Disease development is favored by cool, wet weather with an optimum temperature of about 24°C. Leaf spot is restricted and may disappear at temperatures above 30°C.

Management

Cultural practices — The use of hot- water-treated seed and the production of seedlings in seedbeds where cruciferous plants have not been grown for at least three years will help to prevent introduction of the pathogen to the field. Cauliflower should be transplanted to land where cruciferous plants have not been grown for at least three years. Incorporation of crop residues into the soil as soon as possible after harvest helps to destroy the pathogen and reduces the possibility of disease appearing in the next cauliflower crop. This is especially important where crop rotation is not practiced. If leaf spot is present during harvest, the harvested heads should be precooled rapidly by cold air or crushed ice to prevent further infection of the curd during transit to market.

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(Original by P.D. Hildebrand)

▶ 8.2 Black rot *Figs. 8.2a-f*

Xanthomonas campestris pv. *campestris* (Pammel) Dowson

Black rot is one of the most destructive diseases of cruciferous crops worldwide. The disease usually occurs annually in most crucifer-producing regions and yield and quality losses may be high. All *Brassica* vegetables are susceptible to this disease. Several cruciferous weeds are also hosts of the pathogen.

Symptoms Black rot can appear on plants at any growth stage. On young plants, margins of cotyledons turn black and they may drop off. On true leaves, symptoms appear along leaf margins as yellow, V-shaped lesions, with the base of the V usually directed along a vein *(8.2a).* As the lesion expands toward the base of the leaf, the tissue wilts and eventually becomes necrotic *(8.2b).* The infection may move down the vascular tissue of the petiole and spread up or down the stem of the plant and into the roots. Systemic infection can produce scattered, yellow lesions on leaves anywhere on the plant.

The veins of infected leaves, petioles, stems and roots turn black as the pathogen multiplies, thus impairing the normal flow of water and nutrients *(8.2d).* Severely affected plants may lose a few or many leaves. On root crops, such as rutabaga and radish, foliar symptoms may not be visible, but blackened vascular tissues appear in the edible portion of the plant *(8.2e).* On cabbage and Brussels sprouts, infection may spread through the veins into the main stem and leaves of the head rendering the product unmarketable. Black rot infection is often followed by soft rot organisms that further reduce the quality and storage life of the product.

The presence of black veins in yellow lesions along leaf margins is diagnostic of black rot. When affected stems and petioles are cut crosswise, the vascular ring may appear black. The blackened veins also can be seen when stems are cut lengthwise. These symptoms closely resemble those of fusarium yellows, except that vein discoloration in fusarium yellows is brown. Small areas of infection and discoloration may also occur outside the vascular ring with black rot. A yellow, bacterial-laden ooze may exude from cut vascular tissues.

Causal agent *Xanthomonas campestris* pv. *campestris* is a Gram-negative, non-sporing, aerobic rod, 0.4 to 0.7 by 0.7 to 1.8 pm, with a single polar flagellum. The bacterium produces a mucilaginous, extracellular polysaccharide known as xanthan, which is responsible for plugging the xylem tissue and disrupting the flow of water and nutrients. A yellow, membrane- bound, water-insoluble pigment known as xanthomonadin also is produced and imparts a yellow color to bacterial colonies grown on certain media.

The pathogen produces yellow, convex, mucoid colonies *(8.2f)* on yeast-extract, dextrose or calcium-carbonate agar. Several semi-selective media have been developed for isolating the pathogen from seed and soil.

Disease cycle The pathogen survives on seed, which frequently is the most important source of inoculum for infection in seedling beds. Under field conditions, as few as three infected seeds in 10 000 (0.03%) can cause black rot epidemics. The pathogen also can persist in diseased crop residues in the field for up to two years before the residue is completely decayed. The organism may also survive in soil for about 40 to 60 days.

On newly emerged seedlings, black rot bacteria enter stomata along the margin of cotyledons, where the infected seed coat is often attached. The bacteria then migrate intercellularly until they reach the xylem tissue and from there spread thoughout the plant. On true leaves, the bacteria enter through hydathodes located at the ends of veins along leaf margins *(8.2c)* and from there spread throughout the plant.

The optimum temperature for growth of the pathogen is 25 to 30°C. Under these conditions, symptoms may appear on plants 7 to 14 days after infection. At lower temperatures, symptoms develop more slowly.

The pathogen spreads from infected plants and infested crop refuse and soil by splashing water, wind, insects, machinery and field workers. Long-distance spread is by infested seed and transplants.

Management

Cultural practices — Seed that has been certified as disease-free should be selected whenever possible. A hot-water treatment, if done carefully, is an effective method of eradicating black rot bacteria from seed. Seed of cabbage, broccoli and Brussels sprouts should be treated at 50°C for 25 minutes, while seed of cauliflower, kohlrabi, kale, turnip and rutabaga should be treated for 15 minutes. Seed also can be treated with antibiotics, but since these control only bacterial diseases and are usually phytotoxic, they are not generally used.

Seed should be sown in field or greenhouse seedbeds that have been fumigated or sterilized. If this is uneconomical, growers should use seedbeds in which cruciferous crops have not been grown for at least three years. Seed flats should be sterilized with steam, boiling water or a chemical disinfestant. Seedbeds should not receive run-off water from areas where cruciferous crops have previously been grown. Seeding rates should not be too high, because dense plantings can remain wet for long periods, thus favoring spread and infection by the pathogen.

Temperatures are seldom optimal for symptom expression in the seedbed, so infected plants may inadvertently be transplanted to the field where symptoms can subsequently appear. Seedlings with visible symptoms should be rogued and destroyed, and surrounding seedlings should be observed carefully for possible symptom development. Seedling density should be reduced to allow better drying of the plants. Seedlings should not be trimmed to reduce their growth or to harden them off because the pathogen can be spread by trimming equipment.

Transplanted or direct-seeded crops should be grown where there has not been a cruciferous crop for at least three years. The crop should be kept free of cruciferous

weeds that may also be infected with the pathogen. Plantings should be worked only when the foliage is dry since the pathogen spreads readily under wet conditions. Sprinkler irrigation of diseased crops should be avoided.

If transplants are obtained from a supplier, growers should insist upon certified, disease-free material and documentation that transplants were not trimmed and that only new packaging material was used. Information such as seedlot number and source, dates of pulling and shipping, pest-control schedules, and transit conditions is also useful in judging the health of plant material and helping to identify the source of a disease problem.

Cull piles of rutabaga and other cruciferous crops should not be situated close to production fields or storages. Crop residues should be incorporated into the soil promptly after harvest to speed decomposition and to reduce the risk of pathogen spread. Manure from livestock that have been fed affected culls or land on which affected material has been fed should not be used for the production of cruciferous crops.

Resistant cultivars — Resistant cultivars are available for some types of cruciferous vegetables.

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(Original by P.D. Hildebrand)

▶ 8.3 Head rot *Figs. 8.3a-c*

Erwinia carotovora subsp. *atroseptica* (van Hall) Dye
Erwinia carotovora subsp. *carotovora* (Jones) Bergey *et al.*
Pseudomonas fluorescens Migula
(syn. *Pseudomonas marginalis* (Brown) Stevens)
Pseudomonas viridiflava (Burkh.) Dowson

Head rot is frequently a major constraint to successful production of broccoli in Ontario, Quebec and the Atlantic provinces. Losses can exceed 30% and may be as high as 100%. All of the causal bacteria have host ranges that include several other types of vegetable crops, such as carrot, lettuce and potato.

Symptoms (For symptoms and signs of *Erwinia* spp., see Potato, bacterial soft rot and blackleg, 16.2, 16.3; for those of *Pseudomonas viridiflava,* see Lettuce, pseudomonas diseases, 11.3.) Symptoms first appear after periods of rain when heads have remained wet for several days. Areas on heads colonized by pathogenic strains of *P. fluorescens* appear water-soaked where water has formed in a film *(8.3a).* This is in contrast to unaffected areas, where the waxy surface of the florets imparts a gray-green color and causes the water to form in beads. Small, black lesions frequently develop in association with the stomata within the water-soaked areas of the sepals and pedicels of the florets. The lesions are raised and a dark discoloration, initially associated with the guard cells, spreads to the surrounding tissues. Decay develops on these affected florets or on young, tender florets associated with meristematic areas of the head. During long periods of continuous wetness, decay spreads rapidly and extensively, resulting in sunken areas on the head (*8.3b,c*).

When heads are colonized by saprophytic strains of *P. fluorescens,* water-soaking symptoms appear but heads fail to decay. Similarly, decay remains localized with minimal water soaking when heads are attacked by strains of *P. viridiflava* or *Erwinia* spp., but decay and water soaking may be more extensive when these bacteria are present in combination with saprophytic strains of *P. fluorescens.*

On broccoli florets, symptoms of water-soaking followed by soft decay are diagnostic of this disease when pectolytic strains of *P. fluorescens* are present. Occasionally, colonization of heads by saprophytic strains of *P. fluorescens* may be suspected if heads become water-soaked but fail to decay despite prolonged periods of wetness. If only a few florets are decayed and water soaking is minimal, the presence of wound pathogens such as strains of *P. viridiflava* or *Erwinia* spp. may be suspected.

Causal agents Strains of *Pseudomonas fluorescens* hiovars II and IV, also referred to as *P. marginalis,* have been identified as the primary cause of head rot, but occasionally head rot is caused by interactions of saprophytic- strains of *P. fluorescens* with other soft-rotting bacteria, such as *Erwinia carotovora* subsp. *carotovora, E. carotovora* subsp. *atroseptica,* and *P. viridiflava.*

For detailed descriptions of *E. carotovora* subsp. *atroseptica* and *E. carotovora* subsp. *carotovora,* see Potato, bacterial soft rot, 16.2, and blackleg, 16.3; for *P. fluorescens* and *P. viridiflavida,* see Lettuce, pseudomonas diseases, 11.3. Differentiation among these bacteria is relatively easy based on a few presumptive characteristics. Single bacterial colonies on King's B medium should be obtained from water-soaked or decayed tissue. Fluorescence is determined on King's B medium and presence of pectolytic enzymes is determined on the modified pectate medium of Cuppels and Kelman (see Schaad, Additional references) to which no crystal violet has been added. Biosurfactant activity is detected by mixing colonies on King's B medium in a drop of water with a transfer loop, then transferring the loop to a drop of water on a plastic petri dish. Spreading of the water drop indicates a positive reaction. Pathogenic strains of *P. fluorescens* and *P. viridiflava* fluoresce on King's B medium and cause shallow pits on the pectate medium, but only *P. fluorescens* is biosurfactant positive. Some saprophytic strains of *P. fluorescens* are biosurfactant positive but do not cause pitting. Strains of *Erwinia* spp. are nonfluorescent, do not produce biosurfactant and cause deep pits. Pectolytic activity can also be determined by observing maceration of inoculated potato slices by the various bacterial isolates.

Disease cycle The epidemiology of pathogenic strains of *Pseudomonas fluorescens* is not well understood. The bacterium survives in the soil and may also be present in pond and stream water. During periods of heavy rainfall, the pathogen is splash-dispersed from the soil onto broccoli heads. If the heads remain wet for several days, the bacteria multiply and release a biosurfactant, known as viscosin, and pectolytic enzymes into the drops of water. The biosurfactant reduces the surface tension of water, which then wets the waxy surface of the florets. The biosurfactant enables the bacteria to enter stomata and the surrounding tissues and enhances the action of the pectolytic enzymes. If the heads dry, further ingress of the bacteria and decay do not occur. However, the affected areas on heads are easily rewetted in subsequent wet periods, and decay may develop rapidly if wetness and high temperatures persist for several days.

Head rot develops most rapidly when temperatures are high. The optimum temperature for bacterial growth is 28°C, but growth may occur slowly at temperatures as low as 1 to 2°C. Frost injury in the field and infection by downy mildew may predispose the heads to colonization and decay by the bacteria. Insects such as the tarnished plant bug and flea beetles may cause wounds on the florets that predispose them to infection. Excessive soil nitrogen encourages lush, tender growth that reduces air movement and is very susceptible to bacterial attack. Boron and calcium deficiencies may predispose broccoli heads to infection.

Management

Cultural practices — Production practices that enhance air movement through the crop, such as wider plant spacing and planting rows in the direction of prevailing winds, may be beneficial. Successive plantings should be well away or at least upwind from previous plantings to avoid rain-driven dispersal of inoculum. Excessive application of nitrogen fertilizer should be avoided. Calcium and boron should be maintained at adequate levels.

Heads should be cooled and placed into cold storage soon after harvest. Heads that show signs of severe water soaking in the field are more susceptible to decay in storage.

Resistant cultivars — Cultivars that produce heads above the foliage dry more quickly and appear to be more resistant. Shogun and several breeding lines appear to have some tolerance. No cultivars are immune.

Chemical control — Excessive applications of insecticides or fungicides with surfactants during heading should be avoided since surfactants can enhance bacterial infection.

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(Original by P.D. Hildebrand)

▶ 8.4 Scab *Figs. 8.4a-c*

? *Streptomyces scabies* (Thaxt.) Waksman & Henrici (syn. *Actinomyces scabies* (Thaxt.) Güssow)

Scab occurs on the edible fleshy root or enlarged hypocotyl of radish, rutabaga and turnip. Although the disease does not affect yield, it is economically important because scabs on the roots reduce marketability. The disease is not as severe on root crucifers as on potato, red beet and carrot (see Potato, common scab, 16.5; Beet, scab, 5.1; and Carrot, scab, 6.4). It occurs only sporadically in most regions where these crops are grown.

Symptoms On radish *(8.4c)*, small scale-like spots, each about 1 mm in diameter, appear after hypocotyl enlargement begins. Individual spots may reach 1 to 1.5 cm in diameter. The edges of the spots are raised, while the centers are sunken and pitted. The centers of young scabs initially appear white, but infection by secondary organisms may cause the scabs to discolor and decay.

On rutabaga *(8.4a,b)* and turnip, lesions are circular, often reaching 1 to 1.5 cm in diameter, and are scattered over the surface of the root, or frequently grow together to form a corky band around the root just beneath the soil line. Affected tissues may consist of a tan-colored, superficial or raised layer, or tissues may become pitted and dark following secondary decay. The superficial lesions of scab rarely cause reductions in yield, but even a single lesion is cosmetically unacceptable and may render a root unsaleable.

Causal agent (see Potato, common scab, 16.5) The identification of *Streptomyces scabies* as the cause of scab in crucifers is provisional.

Disease cycle (see Potato, common scab, 16.5)

Management

Cultural practices — (see Potato, common scab) Scab on crucifers is often more severe following a potato crop. Although the pathogen is persistent in field soils, rotation is an important means of reducing inoculum levels. A rotation of potato, grain and one year of hay or another green manure crop, followed by a cruciferous crop, is effective. The green manure crop should be worked down early to facilitate rapid decomposition, and the field kept free of weeds during the fall. Green manure crops encourage the growth of microorganisms that are antagonistic to *Streptomyces scabies.* Fields in sod intended for crucifer production should be worked down early in the year before sowing. Adequate soil moisture levels should be maintained, especially during periods of rapid root growth. However, the use of excessive irrigation over many seasons should be avoided as it may lead to increased levels of other pathogens, such as the clubroot organism.

Resistant cultivars — Some European radish cultivars, such as Sar Katra and Large Scharlakenrode, have partial resistance to scab but are not widely grown in North America.

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(Original by P.D. Hildebrand)

FUNGAL DIESEASES

▶ 8.5 Alternaria diseases *Figs. 8.5a,b*

Black leaf spot
Gray leaf spot

Alternaria brassicae (Berk.) Sacc.
Alternaria brassicicola (Schwein.) Wiltshire
Alternaria raphani Groves & Skolko

Three species of *Alternaria* can cause serious losses in cruciferous crops in Canada. *Alternaria brassicicola* has been detected in 33% of samples of garden crucifer seed in Saskatchewan and cited as a potential threat to the canola (rapeseed) crop in the region. In another study, *Alternaria brassicae* affected 60 to 100% of cabbage plants in several fields in Ontario.

Alternaria brassicae and *A. brassicicola* infect a wide range of cruciferous vegetables, including Brussels sprouts, cabbage, cauliflower, Chinese cabbage, kohlrabi, kale, turnip and rutabaga. *Alternaria raphani* can infect many of these crops but generally it affects only radish.

Symptoms Leaf spotting is the major symptom associated with *Alternaria* infection. Pre- and post-emergence damping-off and damage to the inflorescence of seed crops and to seed can also occur. Pinpoint spots on leaves enlarge to become circular lesions several centimetres in diameter with target-like concentric rings. Lesions are initially yellow-brown and later turn brown to black *(8.5a,b).* Younger leaf tissue is less susceptible to infection than older tissue.

The pathogens sporulate abundantly on foliar lesions and lesion centers may become thin, papery and fall out to give a “shot-hole” appearance. Lesions often grow together leading to large necrotic areas and early leaf drop as the disease progresses. Lesions caused by *A. brassicae* tend to be small and light brown to brownish-gray and are referred to as gray leaf spot. *Alternaria brassicicola* lesions are large, olive-gray to grayish-black and are referred to as black leaf spot. Elongate lesions may occur on petioles, stems and flowers. Small, circular, brown lesions may appear on flower pedicels and calyces. Small dark spots may form on young pods and eventually cause pod distortion and premature shattering in seed crops. In infected maturing pods, the diseased seeds are shrivelled and germinate poorly.

Infected seed may appear healthy with no obvious lesions, although spores may be present on the seed surface and mycelium within the seed. Infected seedlings have circular, somewhat sunken, dark brown to black spots on the cotyledons and dark lesions on the hypocotyls, which reduce plant growth and may resemble the symptoms of wirestem caused by *Rhizoctonia solani.* Infected cauliflower curds have sunken, velvety, dark brown spots with large numbers of spores. Affected broccoli heads have a brown discoloration that begins at the margins of individual flowers and flower clusters. Lesions on cabbage heads placed in storage may enlarge and undergo enhanced rot caused by invasion by secondary fungi and bacteria.

Causal agents *Alternaria* species are unspecialized parasites with simple nutritional requirements and a saprophytic stage outside the host. *Alternaria brassicae* and *A. brassicicola* are not host specific, so crossinfection may occur between different crop types. A resting stage consisting of chlamydospores has been reported for *A. raphani,* and microsclero- tia are known for A. *brassicae.* Although all three fungi can survive in susceptible weeds and perennial crops, diseased residue is a major source of inoculum and a means of perennial perpetuation of these pathogens.

These *Alternaria* species lack a sexual state. The mycelium is branched and septate, and conidia are produced on conidiophores that are generally pale brown or brown and arise separately or in clusters. The conidia are mostly in chains and arise through pores in the conidiophore wall. They are typically ovoid or obclavate with transverse and longitudinal septa. The conidiophores of *A. brassicae* are up to 170 pm long and conidia are brown with beaks up to half as long as the entire conidium. The spore body length is 96 to 114 pm and the spore beak length is 45 to 65 pm. The conidia occur in short chains of two to three, with more cross septa than in conidia of *A. brassicicola. Alternaria brassicicola* produces shorter conidiophores, up to 70 pm long, and dark conidia that, in culture, are generally cylindrical to oblong with a beak that is one-sixth the length of the entire conidium. The spores are generally smaller than those of *A. brassicae***,** measure 45 to 55 pm and are produced in chains of up to 20 or more. They often have a central pore that is readily visible in the cross walls. *Alternaria raphani* resembles *A. brassicae* but it forms chlamydospores, the conidia are in chains of up to six, each measuring 60 to 83 pm, and the conidial beak is intermediate in size, generally 10 to 25 pm in length.

Species of *Alternaria* can be readily isolated from diseased tissue using routine procedures. Cultures of *A. brassicicola* on potato-dextrose agar grow faster and develop a black, sooty color compared to cultures of *A. brassicae, which* have a light-colored mycelium. *Alternaria raphani* does not sporulate as abundantly as the other two species and forms a white, cottony mycelium that also develops many irregular forms, varying from thick, dark, heavily septate mycelium to one with characteristic chlamydospores. On malt agar, the aerial mycelium is cottony and white to deep olive-gray; the submerged mycelium is colorless to olivaceous-black.

Disease cycle Spores are produced in large numbers and may be spread throughout fields by wind and splashing rain or on equipment, humans and livestock. Dissemination also occurs as wind-blown, diseased plant tissue, although the chief means of spread to new fields is through use of infested seed. Transfer of the fungus from infested seedling beds to the field in soil adhering to the roots of transplants is also possible.

Alternaria brassicae and *A. brassicicola* require liquid water and temperatures of 15 and 25 °C, respectively, for 16 hours to initiate infection. Subsequent disease development occurs after two to three days, although alternating wet and dry periods will restrict infection by both species. At least 12 hours of continuous high humidity in excess of 90% RH and temperatures above 14°C are necessary for abundant sporulation. At 10°C, *A. brassicae* produces numerous lesions on host tissue after four days, whereas *A. brassicicola* does not infect host tissues under these conditions.

Management

Cultural practices — Hot-water seed treatment (see black rot, 8.2) reduces or eliminates both internal infection and external infestation of seed by *Alternaria* spp.. Long rotations with non-cruciferous crops, incorporation of diseased crop residues into the soil, elimination of cull piles, eradication of cruciferous weeds, and avoidance of overhead irrigation during head development all will reduce inoculum levels. Seedbeds and successive crops of crucifers should be located away or upwind from existing cruciferous crops to avoid wind-borne inoculum. Also, seedbeds should be kept disease-free to prevent the spread of disease. Control of alternaria leaf spot on cabbage heads in the field is essential if the crop is intended for prolonged storage.

Chemical control — Protective fungicide seed treatments control only fungal spores on the seed surface. Regular applications of protective fungicide sprays beginning in midseason, particularly if conditions are warm and wet, will arrest disease development.

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(Original by R.F. Cerkauskas)

▶ 8.6 Blackleg *Figs. 8.6a-d*

Phoma lingam (Tode:Fr.) Desmaz. (teleomorph *Leptosphaeria maculans* (Desmaz.) Ces. & de Not.)

Blackleg is a destructive fungal disease of many cruciferous crops. Although blackleg can spread rapidly in a field, its progress is not as explosive as that of black rot caused by *Xanthomonas campestris* (see black rot, 8.2).

Blackleg is a serious disease of canola, but has become less important in cruciferous vegetables because of successful disease management practices in the seed industry throughout the world.

Symptoms Plants may become infected in the seedbed or at any time in the field. Symptoms in plants in the seedbed occur two or three weeks before transplanting. Seedling infection is first seen on cotyledons or on the first true leaves *(8.6a).* Seedlings may be killed, while other plants may only have dead, withered cotyledons. Inconspicuous bluish lesions may appear on stems of older plants at the cotyledon scar. Later, an elongate, light brown, sunken area with a purplish or black margin forms on the stem near the soil line. As the lesion gradually extends upward and downward, the stem becomes girdled and blackened. Numerous, small, dot-like, black pycnidia form in these lesions. Stem lesions may extend below the soil line *(8.6b),* causing dark cankers and death of the fibrous roots. Severely infected plants wilt and may lodge. On rutabaga and turnip, cankers form on the fleshy root *(8.6c,d)* and a dry rot may occur in storage. Inconspicuous, circular, light brown to gray spots form on leaves. Large numbers of conidia, often in pinkish gelatinous coils, form in these lesions. Infection of seed pods and seed may occur on crucifer seed crops.

Causal agent Pseudothecia of the sexual state are globose, black, 300 to 500 µm in diameter, with protruding ostioles. Each ascus measures 80 to 125 by 15 to 22 µm, has two distinct walls and contains eight ascospores, each measuring 35 to 70 by 5 to 8 µm. Ascospores are biseriate, cylindrical to ellipsoidal, mostly rounded at their ends, yellow-brown, and guttulate. Pseudoparaphyses are filiform, hyaline, and septate.

Pycnidia of *Phoma lingam* on stems and leaves are of two types: Type I pycnidia are sclerotioid, initially immersed in the host tissue, but eventually erupt, and are variable in shape, 200 to 600 µm across, with narrow ostioles; Type II pycnidia are globose, black, and 200 to 600 µm in diameter.

Conidia are hyaline, short, cylindrical, mostly straight, some curved, guttulate with one guttule at each end of the conidium, unicellular, and 3 to 5 by 1.5 to 2 µm.

The pathogen can be isolated from surface disinfested segments of tissue plated on V-8 agar with 40 µg/mL rose bengal and 100 µg/mL streptomycin. To detect the pathogen in crucifer seed, the seed samples should be placed on moist filter paper and incubated at 20°C in darkness for one day. The seeds should then be transferred to a freezer at -20°C for one day, which prevents seed germination but does not affect the development of the pathogen. The seeds should be incubated further at 20°C with a 12-hour photoperiod of fluorescent light. After 7 to 10 days, pycnidia will develop on infected seeds.

Disease cycle *Phoma lingam* may survive for at least four years in seed and for at least three years in crop residues in the field. The pathogen infects seedlings, causing lesions in which pycnidia are formed. Conidia exude from these pycnidia in long, pink to lilac-colored coils. The conidia are splashed to nearby cruciferous plants, where they germinate and cause new infections. Wet, rainy weather aids in the spread of conidia and reinfection.

The teleomorph state has been found in several areas of Canada, the United States and elsewhere. Ascospores become air-borne and may be transported long distances, causing infections that cannot be traced to infested seed or poor rotation practices.

Management

Cultural practices — Seeds should be hot- water treated as for black rot. A four-year crop rotation should be practiced in seedbeds and fields. Seedbeds should be thoroughly inspected and diseased plants should be rogued. After lifting, transplants should not be sprayed or dipped in water before planting. Cruciferous crops should not be planted adjacent to, or downwind from, fields in which crucifers, including rapeseed/canola, were grown in the previous year, because surface water, wind and rain may spread the fungus from one field to another. Growers should choose fields with good water drainage and air circulation, keep fields free of cruciferous weeds, avoid cultivating plants when they are wet and incorporate crop residues promptly after harvest to speed decay. Manure from animals fed infected plants should not be used as a soil amendment.

Resistant cultivars — Horseradish is resistant to blackleg. Cabbage, Chinese cabbage, Brussels sprouts, most canola and some radish and rutabaga cultivars are susceptible. Cauliflower, broccoli and some canola and turnip cultivars are moderately susceptible. Some turnip, rutabaga, radish and mustard cultivars are only slightly susceptible or immune.

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(Original by R.W. Delbridge)

▶ 8.7 Black root *Fig. 8.7*

Aphanomyces raphani Kendrick

Black root of radish is a minor soil-borne fungal disease that is known to occur in Ontario, Quebec and British Columbia, where losses ranging from 10 to 50% and affecting at least one hectare have been reported. The pathogen attacks radish primarily but also may infect broccoli, Brussels sprouts, cabbage, Chinese cabbage, cauliflower, kale, kohlrabi, rutabaga and turnip.

Symptoms Radish plants are susceptible from the seedling to mature stages, although seedling infection is rarely observed in the field. Symptoms on seedlings consist of dark, water-soaked lesions on the lower hypocotyl. Infected roots, stems, petioles and cotyledons turn black and collapse as the disease develops, often resulting in damping-off. Oospores are present in the invaded tissue, especially in the cortical tissues of the secondary roots in seedlings and older plants, and may even occur in plants without symptoms. In older radish plants, the first symptoms consist of bluish-gray to black areas on the skin where secondary roots, growth cracks or wounds occur. The dark lesions may grow together, resulting in constricted rings around the root and reducing growth. This may lead to deformities and stunting of the tops. The blackening extends deep into the root (*8.7)***.** The rot is dry initially, but as the disease develops, secondary soft-rotting organisms may cause eventual root disintegration. Hypocotyl infection of seedlings of the other crucifers leads to typical damping-off symptoms.

The black or gray discoloration extending deep into the root tissues distinguishes black root from the more superficial, scaly brown lesions associated with rhizoctonia diseases. As well, the mycelium of *Aphanomyces raphani* is non-septate and hyaline.

Causal agent *Aphanomyces raphani* is closely related to *A. cochlioides* (see Beet, aphanomyces root rot, 5.2), and species differentiation is based mainly on oogonial size. The development and life-cycle of the two fungi are similar, although the size and shape of fungal structures differ. The hyphae of *A. raphani* are 8.2 to 11.3 µm in diameter, nonseptate, coarse and profusely branched at right angles, and the oogonia are 32 to 45 µm in diameter. Oospores of *A. raphani* are hyaline and 21.4 to 29 µm in diameter.

Saprophytic bacteria are often associated with diseased host tissue, which therefore should be washed thoroughly and placed in distilled water overnight at room temperature. The identity of the fungus can be verified by zoospore discharge from sporangia produced on pieces of tissue that have been treated in this way. Oospores characteristic of the pathogen are produced in abundance in the secondary roots of radish and other crucifers invaded by the fungus. A partially selective medium containing 150 pg/mL streptomycin sulfate and 10 µg/mL benomyl in radish agar may be used to enhance recovery of *Aphanomyces* spp. from infected plant tissue (see Selected references, Humaydan & Williams). Alternatively, other selective media may be used (see Beet, aphanomyces root rot).

Aphanomyces raphani grows rapidly on artificial media, such as potato- dextrose agar, held at 23°C, often reaching 90 mm in diameter within nine days. Colonies are cream-colored, moist and dense with a tough mycelial mat that is free from aerial hyphae. Cultures are best maintained on radish agar. Abundant oospores and zoospores are produced on this medium. Growth occurs from 12 to 32°C, with the optimum between 18 to 24°C.

Disease cycle The disease is favored by high temperatures, with infection of radish seedlings occurring at 16 to 32°C with a maximum at 27°C. High soil moisture or free water in the soil is necessary for penetration by the motile zoospores. Oospore germination in soil is stimulated by the presence of radish seedlings.

Oospores are produced in large numbers in the secondary roots of many crucifers and in host residues or soil particles that accompany seed. They allow the fungus to persist for long periods and subsequently to infect hosts. Mycelium and zoospores are incapable of prolonged survival in soil but the fungus may subsist as mycelium in volunteer or seed plants. Dissemination is by splashing water or by movement of surface water carrying oospores or zoospores to other plants or neighboring fields. Inoculum dispersal is also possible by infected plants, wind-blown soil or host residue and by tools, agricultural machinery and workers.

Management

Cultural practices — Diseased crop residues should be worked well into the soil and good soil drainage provided. Inoculum levels of *Aphanomyces raphani* in the soil may be reduced by following a four-year rotation with non-cruciferous crops and controlling cruciferous weeds. Some cole crops may escape infection for several weeks after transplanting.

Resistant cultivars — Radish cultivars with resistance to black root include Belle Glade, Fancy Red, French Breakfast and Fuego.

Chemical control — Soil disinfestation with fumigants, although effective, is not economical.

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(Original by R.F. Cerkauskas)

▶ 8.8 Clubroot *Figs. 8.8a-c*

Plasmodiophora brassicae Woronin

Clubroot is a major fungal disease of cruciferous crops worldwide. In Canada, it is a problem primarily in British Columbia and eastern Canada, where entire crops have been destroyed by the disease. Clubroot occurs on broccoli, Brussels sprouts, cabbage, cauliflower, turnip, rutabaga and radish. It can also attack many native and weed species of the mustard family and non-cruciferous genera such as *Agrostis* (bentgrass), *Dactylis* (orchardgrass), *Holcus* (velvetgrass), *Lolium* (ryegrass), *Papaver* (poppy) and *Rumex* (sorrel, dock).

Symptoms The disease can progress considerably before above-ground symptoms are visible. Infected roots enlarge to form galls that vary in size and shape. On crops such as radish, turnip and rutabaga, in which the fleshy root is an enlarged hypocotyl, galls form on the taproot or on secondary roots. These galls are frequently globular or spherical and can be large *(8.8c).* On hosts with fibrous root systems, such as cabbage, cauliflower and broccoli, club-like, spindle- shaped swellings form on individual roots *(8.8b).* These swellings may be isolated and occupy only parts of some roots or they can grow together to occupy the entire root system *(8.8a).* Severely distorted roots are unable to absorb minerals and water. Consequently, top growth is stunted and lower leaves may turn yellow and drop off. Younger foliage may turn bluish and wilt during the day but recover at night. As the disease progresses, the root swellings are often invaded by secondary organisms, causing decay and death of the plant. Seedlings can become infected in seedbeds. Usually, there are no visible symptoms on the top growth, but the tiny, club-like swellings can be seen on the roots. Herbicide injury and hard swellings of unknown origin called hybridization nodules on turnip, rutabaga and rapeseed/canola may be confused with clubroot.

Causal agent *Plasmodiophora brassicae* is a simple fungus that produces a multinucleate mass of protoplasm called a plasmodium, which lacks a cell wall and definite mycelium. Resting spores are uninucleate, hyaline, spherical and measure 2.4 to 3.9 µm. Zoospores are ovoid or variably shaped, biflagellate and 2.5 to 3.5 µm. Zoosporangia average 8 µm. The fungus cannot be cultured *in vitro* but can be grown in callus cultures established from root galls.

Disease cycle In the presence of susceptible roots, resting spores of the fungus germinate to produce motile zoospores that swim in free water and penetrate the surface of the root hairs. The fungus develops into a sporangial plasmodium and subsequently divides into multinucleate portions that develop into zoosporangia. These are released from the host through pores dissolved in the epidermal cell wall, and four to eight secondary zoospores are liberated from each zoosporangium. Some of these zoospores fuse in pairs before germinating and infecting the host.

Plasmodia in the roots cause cells to enlarge abnormally and divide repeatedly, resulting in gall formation. Infected cells are distributed in small groups among healthy cells within the gall. As the plasmodia mature, they transform into masses of resting spores that are released back into the soil when secondary organisms decay the galls. The plasmodium-infected galls use nutrients required by the plant and interfere with the absorption and translocation of nutrients and water in the roots, causing stunting and wilting of the plant. Decay of the galls releases toxic substances that are partly responsible for the wilt symptoms. The fungus invades young root hairs readily, but wounds are necessary for infection of thickened roots and underground stems, although stems also may be invaded through leaf scars.

Plasmodiophora brassicae is disseminated by drainage water, soil that clings to farm equipment, shoes, tools, infected transplants, and contaminated manure and irrigation water. The disease is usually more severe on wet, acidic soils. Repeated crucifer production leads to a rapid build-up of the pathogen. Resting spores may remain viable in the soil for more than 18 years. At least nine pathotypes of the pathogen have been identified. The role of non-cruciferous plants in the epidemiology of the disease is not known.

The optimum mean daily soil temperature for disease development ranges from 19.5 to 23°C. Soil temperatures of at least 16 to 21 °C are required for resting spore germination. Moisture levels above 50 to 70% of the maximum water-holding capacity (about –20 to –15 kPa) of the soil are needed for infection. Clubroot tends to be more severe in soils with a pH of less than 7.0.

Management

Cultural practices — Good soil drainage and the maintenance of a high pH by regular application of lime help to reduce disease incidence. The degree of control may depend on soil pH, which is probably the most important factor influencing disease development. High concentrations of calcium and magnesium may give control below pH 7.2, whereas low calcium and magnesium concentrations may permit disease development above pH 7.2. Calcitic lime is usually more effective than dolomitic lime. If soils are low in magnesium, however, dolomitic lime is preferable. Finely ground lime is more reactive than coarse granules and alters the pH more quickly. Increasing the pH often results in boron deficiency on coarse-textured soils (see boron deficiency, 8.23). Application of boron as a foliar spray or in the transplanting water will alleviate this potential problem.

Long, five- to seven-year crop rotations between cruciferous crops are necessary, and cruciferous weeds must be controlled. The movement of soil or plant material from infested areas into clean fields must be avoided and manure from animals fed infected cull plants or pastured in infected crops should not be used. Transplants should be produced on non-infested soil and the field site should be well- drained and have no history of clubroot. Resting spores of *Plasmodiophora brassicae* may be present in irrigation water and sediment where run-off from infested fields collects in irrigation ponds. Care should be taken to use irrigation water that is not contaminated by the clubroot fungus.

Resistant cultivars — Multi-race resistant cabbage lines and a resistant cabbage cultivar, Richelain, are available. In the Atlantic region, 'Kingston' rutabaga is resistant to all known races and 'York' rutabaga is resistant to several but not all races. The broccoli cultivar Oregon CR1 is resistant to several races but is poor in horticultural quality.

Chemical control — Growers should fumigate seedbeds if disease-free beds are not available. Disease incidence in field plantings can be controlled by using a fungicide in the transplant water.

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(Original by R.W. Delbridge)

▶ 8.9 Damping-off *Fig. 8.9*

Pythium debaryanum of authors, not R. Hesse
Pythium ultimum Trow
Rhizoctonia solani Kühn
(teleomorph *Thanatephorus cucumeris* (A.B. Frank) Donk)

Damping-off can cause serious losses in cruciferous crops in seedling trays in greenhouses *(8.9)* and in direct-seeded plantings in fields. The pathogens causing this disease are common in field soils and can infect many types of vegetable crops.

For information on damping-off, see rhizoctonia diseases in this chapter, 8.13. Also see Celery, damping-off, 7.4.

(Original by P.D. Hildebrand)

▶ 8.10 Downy mildew *Figs. 8.10a-e*

Peronospora parasitica (Pers.:Fr.) Fr.

Downy mildew is a problem on crucifers throughout Canada. Losses in each of broccoli, Brussels sprouts and cauliflower in the lower Fraser Valley, British Columbia, in 1965 and 1966 were estimated at 5 and 2%, respectively. In Ontario, 11 to 30% of cabbage and radish plants in several fields were affected during one survey. In Quebec and Newfoundland, respectively, 10% and 70 to 100% of rutabaga plants in two fields surveyed were affected, while in Nova Scotia 25% of rutabaga plants were diseased.

Downy mildew occurs frequently on broccoli, Brussels sprouts, cabbage, rutabaga, turnip, Chinese cabbage, cauliflower, kale, kohlrabi and radish. Other cruciferous plants such as Arctic draba *(Draba lactea* Adams), dame's-violet *(Hesperis matronalis* L.), false flax *(Camelina microcarpa* Andrz. in western Canada and *Camelina sativa* (L.) Crantz in eastern Canada), garden cress *(Lepidium sativum* L.), gray tansy mustard *(Descurainia richardsonii* (Sweet) O.E. Shulz), pepper grass *(Lepidium densiflorum* Schrad.), scurvy grass *(Cochlearia officinalis* L.), shepherd's-purse *(Capsella bursa-pastoris* (L.) Medic.), tumbling mustard *(Sisymbrium altissimum* L.), wallflower *(Cheiranthus cheiri* L.), and various northern plants also may be hosts for *Peronospora parasitica.*

Symptoms Infection can occur at any stage of growth *(8.10a).* Seedling infection causes the formation of discolored spots on cotyledons. Sporulation of the pathogen may occur on the cotyledon undersurface and hypocotyl. Cotyledons later turn yellow, shrivel and die. At this stage, the fungus is capable of becoming systemic and quiescent.

Initial foliar symptoms consist of discrete, angular, yellow areas on the upper surface of the leaf and fluffy, white, sparse, patchy mycelial growth *(8.10b)* on the undersurface. The affected areas enlarge under moist conditions and turn tan and papery *(8.10c).* In cabbage, systemic invasion of the stem may occur after infection of the lower leaves during the growing season. The fungus may then slowly invade the head leaves and sporulate after the cabbage has been stored. Systemically invaded tissues such as the midrib and blade turn yellowish, then grayish-black and necrotic. Affected cabbage tissue becomes very susceptible to attack by secondary bacteria and fungi. The fungus can cause numerous sunken black spots of varying size on the head.

Cauliflower infection may extend to the head both in the field and in storage. A blackening similar to that observed on cabbage may be evident on curds *(8.10d).* Abundant fungal sporulation and rotting caused by secondary organisms, such as bacteria, often follow the discoloration of cauliflower curds. On broccoli heads, invasion of the fungus cannot be detected by external symptoms. Brown to black streaks may appear in the vascular system of the upper part of the main stalk and branches that lead to the florets. Fungal sporulation followed by rotting occurs on Brussels sprouts as well.

Radish, rutabaga and turnip roots can be invaded systemically, resulting in an irregular, internal brown to black discoloration extending downward from the crown or the soil line. Diseased radish has a brown to black, blotchy epidermal discoloration about half-way down the side of the root *(8.10e).* Often, there is a slight russeting of the epidermal tissue and some cracking. In more advanced stages for all three crops, minute cracks or root splitting may occur. Internal tissue remains firm unless secondary organisms enter and cause decay.

Causal agent *Peronospora parasitica* is an obligate parasite that forms nonseptate mycelium, which occupies the intercellular spaces of the host and forms haustoria within the cells. The hyaline, dichotomously branched sporangiophores emerge through host stomata, bearing hyaline, elliptical, single, terminal sporangia, 16 to 20 by 20 to 22 µm, which germinate by means of a germ tube. Sexual oogonia and antheridia form as the host senesces. The oospore that develops is spherical, 26 to 45 µm in diameter, thick-walled, and yellow-brown. Temperature and relative humidity may influence the morphology of sporangia and sporangiophores, and neither sporangium size and shape nor host range is a reliable character. *Peronospora parasitica* can produce oospores in a range of *Brassica* species and cultivars. However, there is evidence that different isolates consisting of mating types 1 and 2 are required for oospore formation. Evidence suggests that field populations of the fungus are potentially highly variable.

Disease cycle *Peronospora parasitica* exhibits some parasitic specialization at the generic, specific and lower taxonomic levels of the host. For example, turnip isolates may be unable to infect radish or rutabaga. However, the host range of specific isolates of the pathogen from crucifers may be variable and unrelated to the taxonomy of the host family. Also, resistance to the disease increases with the age of the host. Historically, differences in host range were used to delimit distinct species; however, *P. parasitica* is now regarded as a single aggregate species. Although most cruciferous weeds are susceptible to downy mildew, it is not known whether they serve as hosts for the strains of *Peronospora* that occur on cruciferous crops.

Cool, moist conditions favor disease development. Temperatures of 10 to 15°C and abundant moisture on leaves from dew, drizzling rain or heavy fog are optimum for epidemic development. Sporulation, germination and reinfection may occur within four to five days.

The fungus may survive in a latent or quiescent state within systemically infected plants. This may be a source of subsequent lesions on flowering parts like cauliflower curds. The fungus survives between crops as oospores that are formed when the host undergoes senescence. Infection of seedlings in soil contaminated with oospores from decomposed host tissue is possible. Oospores are present in roots and may contaminate seeds also. The survival of sporangia is greatly reduced in dry soil and at low temperatures. Sporangia are disseminated by wind and splashing rain. Mycelium in the seed coats and oospore contamination of seeds are other means of long-distance dispersal.

Management

Cultural practices — Control in the seedbed is very important and includes the use of clean, well-drained soil that has been free from crucifers for the previous two years, avoidance of excessive overhead irrigation to keep seedlings and leaf surfaces as dry as possible, prevention of overcrowding of seedlings and promotion of ventilation within the seedbed by regulation of plant density. Fertilizer can be used to stimulate growth to enable seedlings to outgrow infections. Removal of crop residues from seedbeds should be routine because the oospores can survive in dried foliage.

Resistant cultivars — Several resistant or tolerant hybrid cultivars of broccoli are available. These include Arcadia, Cindy, Citation, Esquire, Eureka, Green Belt, Hi-Caliber, Marathon, Mariner, Pinnacle, Samurai, Sprinter and Zeus.

Chemical control — Preventive spraying in the seedbed with protectant foliar fungicides may be necessary if environmental conditions favor disease development. This may prevent new infections but it will not eradicate established lesions. A regular spray program may be necessary after transplanting or direct seeding in the field if mildew persists.

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(Original by R.F. Cerkauskas)

▶ 8.11 Fusarium wilt (yellows) *Figs. 8.11a,b*

Fusarium oxysporum f. sp. *conglutinans* (Wollenweb.) W.C. Snyder & H.N. Hans.

(syn. *Fusarium conglutinans* Wollenweb.)

Yellows or fusarium wilt was first reported in 1899 on cabbage in New York State. The disease was first observed in Canada in Ontario in 1931. Losses may be significant on a wide range of Cruciferae during warm growing seasons or where resistant cultivars are not commercially available. During extensive surveys of diseases on vegetable crops in Ontario in 1967, the disease was observed in 5 of 24 fields of early heading cabbage, with a range of 10 to 95% of the plants affected.

Cruciferous vegetables that are susceptible to yellows include broccoli, Brussels sprouts, cabbage, Chinese cabbage, cauliflower, collard, kale, kohlrabi and radish. Flowering stock *(Matthiola* spp.) and many native cruciferous plants and weeds are also susceptible.

Symptoms The pathogen can infect plants at any growth stage. The first symptom on cabbage is yellowish-green foliage. Sometimes the yellowing is uniform, but usually it is more intense on one side of the leaf and plant *(8.11a).* The leaf curls or the plant twists when only one side is affected. When the whole plant is affected, the lower leaves first become yellow. This symptom gradually progresses up the plant, which becomes stunted. Affected leaves may drop prematurely. The vascular tissue is yellow at first, then turns brown, dies and becomes brittle. These symptoms may not appear on susceptible plants grown in cool soil in early spring until the soil warms up about the time of crop maturity.

Symptoms of yellows resemble those of black rot and under some conditions may be mistaken for that disease. Both diseases cause curling or dropping of the leaves, drying of the leaf edges, discoloration in the vascular system *(8.11b)*, and finally death of the plant. In black rot, the yellowing of the leaf toward the margin is often in a V-shaped pattern. For yellows, vascular discoloration tends to be yellowish-brown rather than black. Plating of yellows-infected vascular tissue on acid- or antibiotic-amended potato-dextrose agar at 25°C will yield the pathogen within a few days. The black rot bacterium will not grow on the amended agar.

Causal agent *Fusarium oxysporum* f. sp. *conglutinans* is related to, but distinct from, other forms of *F. oxysporum* that cause wilts of solanaceous and legume crops. Four physiologic races of *F. oxysporum* f. sp. *conglutinans* exist. Race 1 is most pathogenic to cabbage, Brussels sprouts, kale, cauliflower and radish, but less so to flowering stock, collard and kohlrabi. Race 2 is most pathogenic to radish, but slightly so or nonpathogenic to Brussels sprouts, kale, flowering stock, cabbage, cauliflower, collard and kohlrabi. Race 3 and race 4 are most pathogenic to flowering stock. Race 4 differs from race 3 in that it causes slight to no wilt in the cultivars Apricot, Double Giant Imperial Rose, Double Early Giant Imperial Golden Rose, Double Giant Imperial Shasta, Standard Gold and Tenweeks White flowering stock.

On potato-dextrose agar at pH 6.5 to 7, *F. oxysporum* f. sp. *conglutinans* colonies may be pale white to cream-colored and the mycelium is floccose throughout. Hyaline microconidia and macroconidia are both borne on phialides. Microconidia are generally abundant, variably cylindrical, oval or straight to curved, one-celled or septate, and measure 2.5 to 4 by 6 to 15 µm. Macroconidia, although sparse in some strains, are generally multicellular, sickle shaped, up to 5.5 µm in width, and 33 µm in length. Hyaline, smooth to rough-walled chlamydospores are generally abundant and may be terminal, intercalary, isolated or in chains.

Disease cycle The pathogen usually infects through seedling rootlets and through roots wounded during transplanting. The fungus moves directly to the water-conducting xylem in the stem and leaves. Part or all of the plant may die. The pathogen then produces conidia and chlamydospores, both inside and outside the affected tissue. The browning of vessels and yellowing of the leaves in advance of the fungus is caused by a toxin, which is produced in the xylem and moves upward in the sap stream.

Yellows is favored by warm weather. It develops to its maximum at 27 to 29°C soil temperatures. It is inhibited above 32°C and does not develop well at soil temperatures below 16°C. Soil moisture and pH have little or no influence on disease development.

The pathogen can survive in soil for a number of years in the absence of a host. Once the pathogen has become established, it can spread rapidly to other areas by wind-borne soil, surface drainage water and soil adhering to farm implements. Long-distance spread is by infested seed or diseased seedlings.

Management

Cultural practices — Extreme care must be taken to avoid using infested seedlings or seed. Once the disease has appeared in an area, the best control strategy is to use yellows-resistant cultivars.

Resistant cultivars — Two types of resistance (A and B) exist in cabbage. Type A cultivars are uniformly resistant at all field soil temperatures; Type B cultivars have some degree of resistance below 21°C. Market Prize, Market Topper and King Cole have Type A resistance. Some cultivars with Type B resistance include Red Hollander, Wisconsin All Seasons and Wisconsin Hollander.

Commercial radish cultivars resistant to yellows are Red King and Fancy Red. Most cultivars of broccoli, Brussels sprouts and cauliflower are resistant to yellows, except in hot weather.

Growers should consult local extension crop specialists regarding cultivars best suited to a particular area.

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(Original by A.A. Reyes)

8.12 Powdery mildew *Fig. 8.12*

Erysiphe polygoni DC. (syn. *Erysiphe cruciferarum* Opiz:Junell)

Powdery mildew is a minor disease of cabbage, cauliflower and the other cruciferous vegetables, except for rutabaga cultivated in southern Ontario in the region bordering Lake Huron, where incidence and severity may be high. *Erysiphe polygoni* has a wide host range.

Symptoms Signs of the disease consist of white, powdery or mealy, superficial, patchy mycelial growth on the upper leaf-surface *(8.12).* The patches grow together as the disease develops, until the entire surface is covered by the fungus. It often spreads to the undersurface of the leaves in the late stages of disease development. Affected leaves successively change color to light green, yellow and tan. The leaves die and abscise in very severe cases, resulting in plant stunting and reduced yield, depending on the stage of growth when infection occurs.

Causal agent The nomenclature of the organism has varied. Previously, the collective name *Erysiphe polygoni* was used for the pathogen of rutabaga, turnip and many other economically important hosts. Recently, some authors have restricted the name to the pathogen affecting the Polygonaceae and used the name *E. cruciferarum* to refer to the pathogen that attacks members of the Cruciferae and Papaveraceae. *Erysiphe polygoni* is an obligate parasite with a wide host range. It forms mycelium that produces numerous haustoria, is white and superficial on the upper leaf- surface, and has a powdery appearance due to the production of numerous, one-celled conidia that are hyaline, narrowly ellipsoid to cylindrical, and measure 24 to 51 by 10 to 17.5 µm. The teleomorph state consists of a dark, completely closed cleistothecium that contains 4 to 10 asci. The cleistothecium also has 10 to 30 characteristically simple, indefinite, mycelioid appendages with lengths one-half to three times the diameter of the cleistothecium.

Disease cycle The fungus occurs in several physiologic races on many plant species. Spores are the chief means of dissemination, being carried long distances by wind to other fields. Survival occurs chiefly in the teleomorph state (cleistothecium), which forms in late summer on the upper leaf-surface of infected plants that remain alive over winter. Limited survival of mycelium in tissue of overwintered plants is possible.

Disease development is favored by low relative humidity, water stress within the host, and the availability of a film of moisture on the leaf surface in which spores can germinate.

Management

Cultural practices — Useful control measures in rutabaga crops include crop rotation, eradication of cruciferous weeds and the destruction of volunteer rutabaga and other crucifers.

Selected references
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(Original by R.F. Cerkauskas)

8.13 Rhizoctonia diseases *Figs. 8.9, 8.13a-e*

Damping-off
Wirestem
Root rot (crater rot)
Bottom rot
Head rot

Rhizoctonia solani Kühn
(teleomorph *Thanatephorus cucumeris* (A.B. Frank) Donk)

Diseases caused by *Rhizoctonia solani* occur wherever cruciferous crops are grown. Depending on the time of infection, this fungus can cause different diseases, such as damping-off, wirestem, bottom rot, head rot and root rot. The pathogen exists in numerous strains that are host specific and exhibit a high degree of variability in virulence toward crops.

Symptoms

Damping-off — Pre-emergence damping-off occurs when seeds decay and fail to germinate or they germinate but the young plants fail to emerge. Loss is often attributed to poor seed. Postemergence damping-off occurs when the stem of the seedling is attacked. Infection usually occurs when seedlings are 2 to 5 cm tall. A water-soaked area completely encircles the tender stem near the surface of the soil. Stem tissues collapse, causing the seedling to wilt and eventually topple over *(8.9).* Damping-off usually develops in foci or along rows within the seedbed or seed flat. Invaded seed serves as a food base, enabling the pathogen to reach adjacent seeds or seedlings. Species of *Pythium* also may be associated with damping-off.

Wirestem — Although seedlings of many non-crucifer- ous crops become increasingly resistant to *Rhizoctonia solani* as they mature, crucifers are frequently attacked even after plants are 10 to 15 cm tall. Wirestem may result from an extension of damping-off, but new infections may occur. The stem darkens above and below the soil line and the outer cortex decays and is sloughed off in sharply defined areas encircling the stem *(8.13a,b).* The stem is wiry and slender at the point of the lesion, hence the name wirestem. Because the stiffened stele of the stem provides support, the plant remains erect but may eventually die when transplanted to the field. If the plant survives, it remains unhealthy, stunted and invariably yields poorly.

Root rot (crater rot) — Turnip, radish, rutabaga and horseradish may be attacked in the field or in storage. The pathogen enters through roots, leaf scars or mechanical injuries. Lesions are usually dark brown, slightly sunken and spongy *(8.13c).* Infected horseradish tissue is light yellow to grayish tan and dry. Infected tissues separate easily from the advancing edge of decay. Secondary soft-rotting bacteria may follow the fungal infection *(8.13d).*

Bottom rot — Cabbage plants can be attacked midway through the season when outer leaves of heads touch moist, infested soil. Brown to black, sunken, elliptical, sharply defined lesions initially appear on the undersides and basal parts of leaves. The lesions may become papery during dry weather. Dark-colored, web-like mycelium may grow on the lesions. Eventually, the lower leaves wilt, turn shades of yellow through black, dry up and may drop off. Plants may recover and produce heads or bottom rot may progress into head rot.

Head rot — During damp weather, bottom rot may evolve into head rot on developing cabbage heads *(8.13e).* The fungus attacks the bases of the wrapper leaves, causing them to drop off, exposing the stem. As the hyphae spread up the stem, the bases of the outer head leaves are attacked and their margins on top of the head turn yellow and dry up. A dark-colored, web-like mycelium may be observed between leaves. The disease may spread over the entire surface of the head and several leaf layers deep. The head remains upright and dark and becomes studded with small brown sclerotia. The decay is initially firm but soft-rotting bacteria may invade, turning affected tissues soft and odorous. The sexual state of the fungus may be observed as a gray or chalk-colored membranous growth on the underside of lower leaves or on the surface of the soil, growing from points of attachment to the stems at the soil line. In storage, lesions established in the field may expand as a firm dark decay (see Lettuce, pseudomonas diseases, 11.3).

The damping-off, wirestem, bottom rot and head rot symptoms are diagnostic of the diseases caused by *R. solani.* It is generally possible to differentiate between postemergence damping-off caused by *R. solani* from that caused by *Pythium* spp. *Rhizoctonia solani* produces stem decay near the soil line and may later advance downward into the roots. *Pythium* spp. generally infect root tips and root hairs and advance upward through the plant to the soil surface. Soil particles usually cling to the coarse mycelium of *R. solani* but not to the fine mycelium of *Pythium* spp. when seedlings are pulled from the soil. The coarse, brown, septate mycelium of *R. solani* may be differentiated microscopically from the fine, hyaline, aseptate mycelium of *Pythium* spp.

In the wirestem phase, mycelium is usually not visible. In the bottom and head rot phases, *R. solani* can be differentiated from *Sclerotinia sclerotiorum.* The decay produced by *R. solani* is initially firm, and small, brown-colored sclerotia may be visible over the decayed surface. This is in contrast to the slimy decay and relatively large black sclerotia produced by *S. sclerotiorum.* On root crops, wefts of cream- to brown-colored mycelium and the typical brown- colored sclerotia distinguish *R. solani* from other root rots.

Causal agent (see Bean, rhizoctonia root rot, 15B.7) Strains of *R. solani* that attack crucifers do not normally infect potato and *vice versa.* Identification of strains is possible by determining pathogenicity on specific hosts and by observing anastomosis between standard tester strains and the strain in question. Strains that attack radish and other crucifers typically belong to anastomosis groups AG-4 and AG-2, respectively.

Disease cycle (see Bean, rhizoctonia root rot, 15B.7; and Lettuce, bottom rot, 11.6) The temperature range for growth of the cabbage strain is 9 to 31°C, with an optimum for infection and disease development of 25 to 27°C.

Management

Cultural practices — To prevent damping-off and wirestem in seedbeds, only sterilized soil or soil that has not previously supported crucifers should be used. Seeds should be treated with hot water and a suitable fungicide. Plant density should permit adequate light penetration and air circulation. Growers should avoid overwatering and reduce or eliminate watering on cloudy days. Water should be applied in the morning so that plants can dry off early in the day. Factors such as deep planting, reduced vigor of seed and excessively cold, hot, moist or saline soils may increase seed decay and pre-emergence damping-off. Deficiencies of calcium, potassium and nitrogen, or excessive nitrogen, may promote disease.

Wirestem on cabbage seedlings may develop into bottom and head rot, so affected seedlings should not be transplanted to the field. A rotation of at least three years with non-cruciferous crops should be practiced. When cultivating, soil should not be mounded or hilled onto lower leaves of plants.

Root crops such as turnip, rutabaga and horseradish with only slight infections may safely be stored at low temperature.

Resistant cultivars — Breeding of cruciferous cultivars resistant to *Rhizoctonia solani* has not been studied extensively because the organism was thought to have a wide host range and differences among isolates were not obvious. With the recognition of host specific strains, the potential for breeding for resistance has improved.

Chemical control — Benches, flats and tools should be sterilized using a suitable disinfestant. Soil may be drenched with fungicides after seeding.

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(Original by P.D. Hildebrand)

▶ 8.14 Sclerotinia rot (cottony soft rot) *Fig. 8.14*

Sclerotinia sclerotiorum (Lib.) de Bary
(syn. *Whetzelinia sclerotiorum* (Lib.) Korf & Dumont)

Sclerotinia sclerotiorum is capable of attacking an exceptionally wide range of vegetable crops (see Lettuce, white mold, 11.9). Among crucifers, cabbage and Brussels sprouts are the most commonly affected. *Sclerotinia minor* Jagger also infects cruciferous crops, but it has not been observed on crucifers in Canada.

Symptoms The first symptoms on cabbage are water- soaked areas on stems and lower leaves, especially those in contact with the soil, and also on upper surfaces of the head. As the lesions expand, the leaves wilt and the fungus may spread to the rest of the plant. Affected tissues turn soft and watery and become covered with white cottony fungal mycelium in which numerous, irregularly shaped sclerotia are embedded *(8.14).* The sclerotia are initially white but later turn black. The fungus may spread rapidly from infected to healthy heads within bins during transit or in storage if low temperatures are not maintained.

Stored cabbage can also be attacked by *Botrytis cinerea* (see Lettuce, gray mold, 1 1.10). Gray mold lesions are water-soaked, gray-green and often covered with masses of powdery gray spores, which readily distinguish this disease from sclerotinia rot. Gray mold usually attacks cabbage leaves late in the storage period.

Sclerotinia rot is primarily a storage disease of rutabaga and turnip. After initial infection, a slightly pinkish color may be present on the margin of a lesion, while the inner portion of the lesion is pale brown and water-soaked. The typical white, cottony mycelium and sclerotia appear later on infected tissues.

Causal agent (see Carrot, sclerotinia rot, 6.15)

D**isease cycle** (see Carrot, sclerotinia rot) The pathogen may slowly continue to colonize tissues of field-grown cabbage, rutabaga and turnip placed in cold storage, resulting in pockets of decayed plant material within the bins. The fungus may also survive on infected residue adhering to wooden storage bins, which may act as a source of inoculum.

Management

Cultural practices — Once *Sclerotinia sclerotiorum* has become established in a field, it is difficult to destroy all sclerotia. Soil tillage usually brings enough sclerotia to the soil surface to initiate disease if environmental conditions are favorable. Rotation with non-susceptible crops reduces the number of viable sclerotia. A three- year period with crops such as corn, cereals or grasses is recommended. Susceptible crops should be planted on well-drained soils. Many weed species are susceptible to *S. sclerotiorum,* so fields should be kept weed-free. Pathogens that cause necrotic lesions should be controlled and wounding during harvest should be avoided. Harvested produce should be placed into clean bins for storage. Removal of soil from rutabaga and turnip roots by washing will reduce disease development in storage. Proper temperature and ventilation should be maintained during storage.

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(Original by P.D. Hildebrand)

▶ 8.15 White rust *Fig. 8.15*

Albugo candida (Pers.) Kuntze (syn. *Albugo cruciferarum* (DC.) S.F. Gray)

White rust is of minor importance to radish root crops, but losses to radish seed crops may be severe because of distortion and destruction of flower parts. *Albugo candida* occurs on many cruciferous plants, but yield and quality losses usually are minor, except for turnip rape (*Brassica campestris* L.), in which damage may be significant. The pathogen exhibits a high degree of host specificity, so infection of radish from overwintering inoculum on cruciferous weeds or crops, except canola, is unlikely.

Symptoms Infection may be local or systemic. Local infections, consisting of white, shiny, raised pustules or sori *(8.15)*, develop on upper and lower leaf-surfaces and on stems. The pustules arise from masses of sporangia, which form under the leaf epidermis. When the pustules rupture, the powdery, dry sporangia become wind-borne. Systemic infection of young stems and flowering parts causes enlarged and abnormal development of sepals, petals, pistils and anthers and prevents normal radish seed development. The infected flowers, flower stalks and seed pods become enlarged and develop into distorted, staghead galls, which contain many oospores. The stagheads initially are green, but become brown and brittle at maturity. The fungus often coinfects the same tissues, especially the distorted flower parts, with the downy mildew fungus *Peronospora parasitica.*

Causal agent *Albugo candida* is an obligate parasite that forms mycelium in host tissue that is nonseptate and intercellular with knob-like haus- toria. Club-shaped sporangiophores are produced from the mycelium in a layer beneath the host epidermis. Sporangia are produced in chains from the sporangiophores, with the oldest at the tip of the chain, and become readily detached at maturity. The sporangia are hyaline, nearly spherical, and 14 to 16 by 16 to 20 µm. Germination is primarily by production of zoospores that contain a disk-like vacuole on one side. Oogonia and antheridia are formed from mycelium in the intercellular spaces of the host tissue, especially in systemic infections. The oospores that form have a warty wall, which is a useful trait for distinguishing species of *Albugo*. The oospores are chocolate-colored, 40 to 55 µm in diameter and usually are found in the stems and seed.

Disease cycle The fungus overwinters as thick-walled oospores inside the staghead galls or as mycelium in living tissues. The stagheads may break off the plant and fall to the ground, eventually releasing the oospores. The oospores may germinate and infect the cotyledons and leaves of young plants in the spring. Pustules that develop on leaf surfaces contain many sporangia that are released and dispersed chiefly by wind but also by rain and insects, to neighboring plants. Several generations of sporangia are produced on plants during a growing season. Seed-borne oospores are another important source of inoculum. Under dry conditions in a laboratory, oospores have germinated after 17 years of storage; however, their longevity in natural soils has not been determined.

Disease development is favored by moist conditions and temperatures between 10 and 25°C. Chilling of the sporangia is necessary to initiate zoospore production. Germination occurs over a range of 1 to 20°C and is optimal between 10 and 14°C. Temperatures above 25°C reduce the rate and amount of release of zoospores. The motile zoospores swim about for a short time, then produce germ tubes that invade the host through stomata. Moisture on the host surface is essential for germination and infection.

Management

Cultural practices — The incorporation of infested crop residues into the soil helps to reduce levels of pathogen inoculum. Radish crops should be planted some distance from where they were grown previously. Volunteer rapeseed and wild mustard plants are a source of inoculum and should be eradicated early in the growing season. During the development of the flowering crop, furrow irrigation should be practiced instead of overhead irrigation, because it is less likely to spread the pathogen.

Resistant cultivars — The radish cultivars Chinese Rose Winter, Round Black Spanish, and Burpee White exhibit some resistance to white rust.

Chemical control — Seed treatment with broad-spectrum fungicides is effective in minimizing spread through contaminated seed.

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(Original by R.F. Cerkauskas)

VIRAL DISEASES

▶ 8.16 Turnip mosaic *Figs. 8.16a-c*

Turnip mosaic virus

Turnip mosaic is a widespread, destructive disease of rutabaga in south-central Ontario, where most of the rutabaga production in North America occurs. The host range of turnip mosaic virus is wide and potential sources of inoculum include many cruciferous vegetables and weeds. Losses can be devastating during the growing season in Oriental crucifers, horseradish, lettuce, spinach and mustard crops. Infected rutabaga roots are susceptible to breakdown in storage.

Symptoms The first indication of turnip mosaic infection in rutabaga is premature yellowing of the basal leaves, affecting groups of plants within the field. In severe infections, the entire crop may exhibit yellowing *{8.16a).* The younger leaves become distorted and may assume a wrinkled or blistered appearance with severe light and dark green mosaic-mottling *(8.16b).* Vein banding and veinal flecking may develop on the apical leaves. Pale chlorotic and necrotic lesions are often present on the basal leaves. Older leaves senesce quickly. As new leaves are produced, older ones drop off when mature, resulting in a “goose-necked” appearance. When plants are infected early in their development, roots are severely stunted *(8.16c)* and loss of leaves makes mechanical harvesting difficult. In southern Ontario, disease symptoms are generally evident three weeks after the plants become infected, but symptoms vary depending on the cultivar, the stage of plant development when infection occurs, and the environmental conditions after infection.

Recognition of turnip mosaic in the field is based on foliar distortion and mottling, particularly on younger leaves. Leaf samples should be sent to a diagnostic clinic for positive identification.

Causal agent Turnip mosaic virus is a flexuous filament, approximately 720 nm in length, containing a single linear molecule of RNA. It is sap transmissible to dicotyledonous plants in many families, and it is transmitted by many aphid species in a non-persistent manner.

Turnip mosaic virus occurs worldwide and is frequently reported in the temperate zone of North America. In Ontario, four strains of the virus have been identified, the most common one being identical to a crucifer- infecting strain in New York State. The remaining strains of turnip mosaic virus have narrow host ranges and are limited to a few *Brassica* crops and cruciferous weeds. Only one of the strains is important on rutabaga.

Disease cycle Turnip mosaic virus is not seed-borne. Until recently, the major source of virus has been limited to infected rutabagas, either volunteers or those dumped from storage during early spring. Other reservoir plants are limited to a few weeds in the mustard family.

Natural spread of turnip mosaic virus in the field is only by aphids (see aphids, 8.39). In general, early plantings of rutabaga that become infected provide a source of inoculum for outlying rutabaga fields. Aphid transmission from rutabaga to newly planted winter rapeseed occurs in the fall, allowing for overwintering of the virus.

In 1985, an epidemic of turnip mosaic virus in southern Ontario caused more than 30% loss in the rutabaga crop. The outbreak coincided with an increase in winter rapeseed production near rutabaga fields, which began in the early 1980s. The effect of the virus on rapeseed yield is minimal but losses to rutabaga have increased each year. Infection of rutabaga fields several kilometres distant from the nearest source of inoculum is caused by movement of virulifer- ous aphids from winter rapeseed to rutabaga in early July. This disease has been particularly serious on the major rutabaga cultivar Laurentian.

Management Control of turnip mosaic virus depends on timely control of the aphid vectors (see aphids, 8.39), because symptoms may not appear until long after the aphids have disappeared.

Cultural practices — Volunteer rutabaga plants should be disked in the fall and left on the surface to freeze. In the spring, volunteer plants should be eliminated and culls from storage should be worked into the soil to enhance decomposition. Eradication of mustard and volunteer rape- seed in or around rutabaga plantings helps to reduce infection. If possible, rutabaga should be grown in isolation from other cruciferous crops, in particular winter and spring rapeseed and spring canola. Early season planting allows rutabaga roots to size up before the introduction of virus by aphids, the benefit being that late-infected crops produce a marketable root with better storage success. In southern Ontario, rutabagas should be sown preferably before mid-June. Rutabagas with a high level of virus infection should be marketed early to avoid storage losses.

Resistant cultivars — Sources of resistant germplasm are being developed.

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(Original by L.W. Stobbs)

NON-INFECTIOUS DISEASES

▶ 8.17 Black speck of cauliflower *Fig. 8.17*

Black speck is a minor physiological disorder of cauliflower, occurring mainly on North American Snowball cultivars and rarely on cultivars originating in Europe, Japan or Australia. The cause of black speck is unknown, but it may be a nutrient deficiency. Symptoms appear as black necrotic specks only on branches or flower stalks in the interior of the curd. Several layers of cells collapse and become discolored, resulting in slightly sunken black lesions *(8.17).* The lesions may range from 0.5 to 4.0 mm in diameter. Specific control measures are not available. Black speck of cabbage is a storage disorder.

Selected references

Loughton, A., and J.W. Riekels. 1988. Black speck in cauliflower. *Can. J. Plant Sei.* 68:291-294.

(Original by P.D. Hildebrand)

▶ 8.18 Brown bead *Fig. 8.18*

Brown bead is a physiological disorder of broccoli that occurs sporadically and is usually associated with rapid growth during periods of high temperature following periods of abundant rainfall. The disorder may cause significant losses. Symptoms appear only on the broccoli heads as they approach maturity. Floral buds turn tan or brown *(8.18)* and become easily detached. Adequate supplies of well-rotted organic matter, particularly on light soils, help to prevent surges in growth by maintaining a constant supply of water and nutrients. Some cultivars appear to be less susceptible than others.

Selected references

Flint, M.L., ed. 1985. *Integrated Pest Management for Cole Crops and Lettuce.* Univ. Calif., Statewide Integrated Pest Management Project, Div. Agric. Nat. Res., Oakland. 112 pp.

(Original by P.D. Hildebrand)

▶ 8.19 Growth cracks *Fig. 8.19*

Growth cracking is a physiological disorder that affects the developing hypocotyls and roots of rutabaga and turnip and heads of cabbage. Marketable yields are usually affected to some degree in most crops.

Symptoms Cracks originate in the neck region and may extend down the side of the root *(8.19).* The exposed tissue may be colonized by soft-rotting bacteria, especially *Envinia* spp., which transform the entire root into a jelly- like mass. The interior of the root may be completely decayed, leaving a shell of outer tissues. Initially there is no odor but the tissue becomes putrid following bacterial decay. Growth cracking also can occur in cabbage heads.

Causal agent Growth cracks appear during periods of rapid growth, especially when heavy rainfall or irrigation follows a dry spell.

Management

Cultural practices — Factors affecting rapid growth must be controlled. Uneven spacing, which allows rapid growth of some roots within the row, can be avoided by precision seeding. Excessive fertilization, especially with nitrogen, and manuring should be avoided. Adequate levels of well-rotted organic matter, particularly on light soils, help to prevent surges in growth by maintaining a constant supply of water and nutrients. Decay caused by secondary organisms is also aggravated by warm, humid weather and dense plant canopies. Maintaining soil moisture at uniform levels reduces the incidence of growth cracking. On a small scale, cabbage plants can be given a half twist to break off some of the roots. This will limit water uptake and slow down the growth until the heads can be harvested. Affected roots and heads should not be stored because they are susceptible to bacterial decay and may act as a source of decay within storage bins.

(Original by R.W. Delbridge)

▶ 8.20 Hollow stem *Fig. 8.20*

Hollow stem is a common physiological disorder of broccoli and occasionally cauliflower.

Symptoms Symptoms occur internally in stems and are not usually visible externally. Small elliptical cracks develop in the stem. As the plant approaches maturity, the cracks may enlarge and grow together, causing the stem to become hollow *(8.20).* In severe cases, the hollow area may extend into the floret region of the head. Tissues within the hollow areas are usually not discolored, but tissue discoloration and breakdown may develop soon after harvest.

Causal agent Although the symptoms are similar to those of boron deficiency in cauliflower, the role of boron in hollow stem is not clear. A balance between boron and nitrogen levels in tissues appears to be important, but this relationship has not been clearly defined. Calcium may also have a role in this disorder. Factors that favor rapid plant growth after initiation of the head tend to promote hollow stem.

Management

Cultural practices — Methods that maintain an even rate of growth should be followed. Growers should avoid excessive nitrogen fertilization, especially after head initiation. A uniform, close plant spacing will help to maintain an even growth rate and greatly reduces the incidence of hollow stem. However, at too high a plant density, head size can become too small. Harvested broccoli should be cooled immediately to inhibit bacterial activity in the stem cavities.

Resistant cultivars — Variable susceptibility to this disorder exists among cultivars, but no cultivar is totally resistant. Many of the recently developed hybrid cultivars of cauliflower are very susceptible to hollow stem.

Selected references

Scaife, A., and D.C.E. Wurr. 1990. Effects of nitrogen and irrigation on hollow stem of cauliflower *(Brassica oleracea* var. *botrytis). J. Horde. Sci.* 65:25-29.

Shattuck, V.I., and B.J. Shelp. 1987. Effect of boron nutrition on hollow stem in broccoli (*Brassica oleracea* var. *Italica). Can. J. Plant Sci.* 67:1221-1225.

Shattuck, V.I., B.J. Shelp, A. Loughton and R. Baker. 1986. Environmental stability of yield and hollow stem in broccoli (*Brassica oleracea* var. *Italica). Can. J. Plant Sci.* 66:683-688.

Tremblay, N. 1989. Effect of nitrogen sources and rates on yield and hollow stem development in broccoli. *Can. J. Plant Sci.* 69:1049-1053.

Vigier, B., and J.A. Cutcliffe. 1984. Effect of boron and nitrogen on the incidence of hollow stem in broccoli. *Acta Horde.* 157:303-308.

(Original by P.D. Hildebrand)

▶ 8.21 Intumescence (edema, enation, neoplasm, thrips pustule) *Figs. 8.21 a-**c***

Intumescence is a physiological disorder that can affect the leaves of most crucifers, but it is of most concern when it affects the leaves of cabbage heads.

Symptoms Intumescence is characterized by small, wartlike protuberances that develop on either side of the first 3 to 10 outer leaves of the cabbage head *(8.21a).* The protuberances develop in varying densities and may grow together to form irregularly-shaped, elevated areas. The epidermis may split as the inner cells of the leaf enlarge, subdivide and push outwards *(8.21b).* The exposed cells are initially white and give the protuberances a crystalline appearance. Later, these tissues turn brown and become corky. The margins of severely affected leaves may also dry to a thin, papery texture during storage. Losses due to trimming of affected leaves may be significant.

Causal agent The disorder follows days when the soil is warm and wet and the night air cool and saturated with water. Under these conditions, water uptake by the roots occurs more rapidly than loss by transpiration. This stimulates cell enlargement and division in the hypodermis, resulting in sufficient pressure to rupture the epidermis. The disorder may also be aggravated by injury of the epidermis due to drifting sand, thrips feeding *(8.21c)***,** herbicide residues and air pollution.

Management

Cultural practices — Growers should avoid excessive irrigation during periods when day-to-night temperatures vary greatly. Using windbreaks to reduce soil erosion and controlling thrips may help prevent the problem.

Selected references

Sherf, A.F., and A.A. MacNab. 1986. *Vegetable Diseases and Their Control.* 2nd ed. J. Wiley & Sons, New York. 728 pp.

(Original by L.S. Bérard)

▶ 8.22 Tipburn, internal browning *Figs. 8.22a,b*

Tipburn and internal browning are physiological disorders of similar origin. Chinese cabbage is particularly prone to this disorder, which also affects Brussels sprouts, cabbage and cauliflower.

Symptoms The inner leaves of heads of cabbage and Brussels sprouts are affected, but there are no external symptoms. Margins of inner leaves turn brown, beginning at the hydathodes, and later desiccate to become papery at the margin or over large portions of the leaf *(8.22a,b).* The affected tissue may turn dark brown to black, occasionally being invaded by secondary bacteria that cause a watery soft rot. When tipburn occurs in stored cabbage, symptoms may not be noticed until heads are spot checked before marketing. Brussels sprouts are usually sampled before processing. If the incidence of internal browning is high, the shipment may be rejected. In cauliflower, internal leaves turn brown and fold over the developing curds. When secondary microorganisms attack these leaves, they become mushy, smear over the curd and render it unmarketable.

Causal agent Tipburn and internal browning are caused by inadequate transport of calcium to rapidly growing tissues. Low levels of calcium at the leaf margin result in tissue collapse. Excess nitrogen results in large outer leaves that accumulate calcium at the expense of young expanding leaves within heads.

Environmental conditions that favor rapid plant growth also favor tipburn. Abundant soil moisture promotes rapid growth, while excess moisture reduces soil oxygen levels, which in turn reduces calcium uptake and movement. A dry spell after a period of abundant moisture may aggravate the disorder. Cruciferous crops grown on sandy soil are usually more prone to tipburn compared to plants grown on heavier-textured soils.

Management

Cultural practices — Factors that promote rapid plant growth should be avoided. Maintenance of optimum fertility is important. Maintaining a phosphorus to potassium ratio of 1:1 should help to minimize the incidence of tipburn. Irrigation may be necessary to maintain optimum levels of soil moisture. Addition of high levels of calcium to the soil and foliar applications do not seem to alleviate the problem. Close plant spacing and prompt harvesting of crops when mature are beneficial practices.

Resistant cultivars — Cultivars that grow less vigorously are less prone to this disorder. Resistant cultivars of Brussels sprouts and cabbage are available.

Selected references

Maynard, D.N., and A.V. Barker. 1972. Internal browning of Brussels sprouts: a calcium deficiency disorder. *J. Am. Soc. Hortic. Sei.* 97:789-792.

Maynard, D.N., B. Gersten and H.F. Vernell. 1965. The distribution of calcium as related to internal tipburn, variety, and calcium nutrition in cabbage. *Proc. Am. Soc. Hortic. Sei.* 86:392-396.

Palzkill, D.A., T.W. Tibbitts and P.H. Williams. 1976. Enhancement of calcium transport to inner leaves of cabbage for prevention of tipburn. *J. Am. Soc. Hortic. Sei.* 101:645-648.

Rosen, J. 1990. Leaf tipburn in cauliflower as affected by cultivar, calcium sprays, and nitrogen nutrition. *HortScience* 25:660-663.

(Original by P.D. Hildebrand and L.S. Bérard)

NUTRITIONAL DISORDERS

Despite optimum fertilization with nitrogen, phosphorus and potassium, crucifers sometimes show other nutrient deficiency symptoms. Nutrients may become unavailable in soils that are alkaline or excessively acidic. Soil in which crucifers are grown should be maintained at pH 5.8 to 6.5 for optimum growth.

▶ 8.23 Boron deficiency (brown heart, mottled heart, raan, water core) *Figs. 8.23a-d*

Crucifers, especially cauliflower, rutabaga and turnip, are sensitive to boron deficiency. Plants require boron for translocation of carbohydrates and regulation of plant growth hormones. Boron deficiency results in poorly developed cell walls that collapse.

Symptoms The first visible symptom in cauliflower appears on the head as a firm, tan-colored or water-soaked spot *(8.23a).* Water-soaked areas of internal stem tissues may also occur. The discoloration usually darkens and may spread over the entire head, but the curd remains firm

(8.23b). External stem tissues near the base of the midrib of petioles closest to the head may crack, become corky and turn brown. Cavities that eventually form in stem tissues also turn brown, and the tips of the youngest leaves become light brown. The curd acquires a bitter taste.

In rutabaga and turnip, boron deficiency occurs in the edible root and first appears as areas of brown discoloration that are scattered, grouped or arranged in a concentric pattern *(8.23c).* Discoloration is usually more pronounced in the central area of the root. Symptoms are usually restricted to the lower two-thirds of the root, but in severe cases they can extend from the bottom to the crown, where cavities may form *(8.23d).* Affected tissues become fibrous or punky and may develop a bitter flavor. They also may be invaded by secondary soft-rotting organisms. Roots that are mildly affected usually lack external symptoms. In severe cases, the roots are reduced in size and external root tissues may become rough, corky or leathery. Leaf margins of severely affected plants are typically chlorotic, and a purplish tinge may develop on the underside of the leaves.

Causal agent Boron deficiency occurs most commonly on soils that are coarse or sandy and subject to excessive leaching, resulting in soluble boron levels of less than 0.5 ppm, or on soils with a pH greater than 7. Boron also becomes less available during long periods of drought.

Management

Cultural practices — Irrigation may help to prevent boron deficiency by maintaining a uniform soil moisture. On soils deficient in boron, a boronated fertilizer is required. Only foliar sprays of boron should be used on high pH soils. Application of boron after the appearance of deficiency symptoms is usually too late to correct the problem.

Resistant cultivars — Variable susceptibility to this disorder is expressed in some cultivars. No cultivar is totally resistant.

Selected references

Cutcliffe, J.A., and U.C. Gupta. 1987. Effects of foliar sprays of boron applied at different stages of growth on incidence of brown-heart in rutabagas. *Can. J. Soil Sci.* 67:705-708.

Gupta, U.C. 1979. Boron nutrition of crops. *Adv. Agron.* 31:273-303.

Shattuck, V.I., and B.J. Shelp. 1985. Brown heart in rutabaga. Ontario Ministry Agric. Food. *Factsheet.* 2 pp.

(Original by P.D. Hildebrand)

8.24 Magnesium deficiency *Fig. 8.24*

Broccoli, cabbage, cauliflower and kale are the cruciferous crops that are most sensitive to magnesium deficiency. Brussels sprouts and turnip are much less affected.

Symptoms Symptoms of magnesium deficiency first appear on the older leaves as blotches of interveinal chlorosis *(8.24)*. The chlorosis intensifies and may be accompanied by interveinal purple blotches, especially near the leaf margins. As the plant matures, these symptoms progress up the plant. Orange and red tints also may occur, especially on the leaf undersides. Under severe magnesium deficiency, only the youngest leaves remain green.

Causal agent Magnesium is required for the production of chlorophyll in plants. Deficiencies commonly occur on sandy, acidic soils.

Management

Cultural practices — Dolomitic limestone, which consists of calcium and magnesium carbonate, can be applied to correct this problem. This form of limestone simultaneously raises the soil pH. Magnesium may also be applied with fertilizer, or plants may be sprayed with epsom salts (magnesium sulfate).

Selected references

Scaife, A., and M. Turner. 1983. *Diagnosis of Mineral Disorders in Plants.* Vol. 2. *Vegetables.* H.M. Stationery Office, London. 95 pp.

(Original by P.D. Hildebrand)

8.25 Molybdenum deficiency (whiptail) *Figs. 8.25a,b*

Among cruciferous crops, cauliflower is the most sensitive to molybdenum deficiency, but symptoms may also appear on broccoli, Brussels sprouts and cabbage.

Symptoms In the seedbed, molybdenum deficiency symptoms appear on leaves as small, dark flecks that may have yellow halos. On young plants in the field, molybdenum deficiency symptoms are most common under cool, dry conditions and may appear as areas of interveinal chlorosis, which later become puckered. The chlorotic areas may also develop a purple discoloration. Similar symptoms occur on older plants, particularly along leaf margins, which become thick and brittle *(8.25b)*. The chlorotic areas become necrotic with purple borders, and margins of leaves turn upward, resulting in a cup-shaped leaf. Severe molybdenum deficiency prevents development of the leaf blade, leaving only the midrib with fringes of tissue on either side, resulting in a "whiptail" symptom *(8.25a)*. This symptom may be confused with feeding damage caused by certain insect larvae.

Causal agent Molybdenum is necessary for chloroplast maintenance and nitrate reduction in plants. Molybdenum availability is greatly reduced under acidic conditions, for example, a pH less than 6.5.

Management

Cultural practices — Soils should be maintained at or slightly above pH 6.5. Molybdenum in the form of sodium molybdonate may be applied as a seed treatment, as a foliar spray to transplants before field setting, in the transplant water, as a foliar application in the field, or mixed with fertilizer applied to the soil. Foliar sprays can dramatically correct a deficiency and cause immediate recovery and regrowth. However, if the "whiptail" symptom is present, the problem may not be correctable during the current growing season.

Resistant cultivars — Sensitivity to molybdenum deficiency is variable among cultivars.

Selected references

Hewitt, E.J., and S.C. Argawala. 1951. Production of "whiptail" in cauliflower grown in sand culture. *Nature (Lond.).* 167:733.

(Original by P.D. Hildebrand)

8.26 Sulfur deficiency *Fig. 8.26*

Most cruciferous crops are sensitive to sulfur deficiency.

Symptoms Early symptoms of sulfur deficiency appear as diffuse blotches of interveinal chlorosis on the youngest leaves *(8.26)*. The leaves may also become reflexed, in contrast to the cupping symptom observed in molybdenum deficiency. As the leaves mature, the chlorotic areas may dry to a tan-colored, paper-thin texture.

Causal agent Sulfur is required for protein synthesis and stabilization of chlorophyll in plants. Sulfur deficiency usually occurs in plants grown on soils low in organic matter, particularly in areas far from industrial sulfur dioxide pollution.

Management

Cultural practices — Low levels of sulfur in the soil may be corrected by applying sulfur-containing fertilizers or gypsum (calcium sulfate).

Selected references
Scaife, A., and M. Turner. 1983. *Diagnosis of Mineral Disorders in Plants.* Vol. 2. *Vegetables.* H.M. Stationery Office, London. 95 pp.
(Original by P.D. Hildebrand)

STORAGE DISORDERS

Physiological disorders of stored cabbage are not easily diagnosed because the causes are not always clear. General senescence is one such disorder. Others may appear early in the storage period or while cabbage is still in the field, and their development may be altered by frost injury. Several disorders characterized by spotting of varying size and intensity may be ascribed incorrectly to plant pathogens.

▶ 8.27 Black midrib *Fig. 8.27*

Black midrib can be found on cabbage at harvest, but usually it develops soon after the heads are placed into storage.

Symptoms Black midrib initially appears as spots with ill-defined borders in the parenchyma tissue at the base of the midrib on the convex surface of the outer head-leaves. A continuous dark discoloration may subsequently extend more than 10 cm up the midrib *(8.27)*. When the disorder is severe, the epidermis and underlying parenchyma collapse, creating a large, sunken lesion on the midrib. Black midrib occasionally affects only the middle leaves of the head. Symptoms do not extend past the abscission line between the leaf and the main stem.

Causal agent The exact cause of black midrib is unknown. The variable susceptibilty of cultivars suggests a genetic basis for this disorder, and the sporadic occurrence, from year to year, also suggests involvement of environmental factors.

Black midrib has been associated with phenol deposition on cell walls and a potassium content of less than 1 % of the dry matter in the midrib tissue at harvest. Excessive nitrogen fertilization promotes disease development. Exposure to frost in the field may reduce the severity of symptoms. Controlled atmosphere storage may promote symptoms on the middle leaves.

Management

Cultural practices — Growers should maintain proper nutrient levels in the soil, harvest cabbage after a few light frosts, avoid controlled atmosphere storage if susceptible cultivars are grown, trim the affected outer head-leaves, and spot check for symptoms on middle leaves of heads.

Resistant cultivars — Bartolo, Hidena, Decema Extra, Houston Evergreen, Polinius and Slawdena are tolerant to black midrib.

Selected references
Bérard, L.S., B. Vigier and M.A. Dubuc-Lebreux. 1986. Effects of cultivar and controlled atmosphere storage on the incidence of black midrib and necrotic spot in winter cabbage. *Phytoprotection* 67:63-73.
(Original by L.S. Bérard)

▶ 8.28 Black speck of cabbage (pepper spot, spotted necrosis) *Figs. 8.28a,b*

Black speck is a storage disorder of cabbage that occasionally causes significant losses. It is distinct from peppery leaf spot, a bacterial disease that can affect several cruciferous crops, and black speck of cauliflower, which is a physiological disorder.

Symptoms Two types of black speck are recognized on cabbage. Type I affects the green, outer leaves at harvest or in early storage *(8.28a)*. Type II, also known as senescent black speck, affects the pale yellow inner leaves after cabbage has aged in storage *(8.28b)*. Black speck is characterized by the collapse and darkening of guard and adjacent epidermal cells of the stomata, causing scattered, black, sharply sunken, pin-point spots less than 1 mm in diameter on both sides of the cabbage head-leaves.

Causal agent The cause of black speck is unknown. Low storage temperatures may promote the symptoms. Black speck has also been associated with high soil salinity promoted by irrigation during periods of high evapo-transpiration. An imbalance of minerals in tissues has been implicated, especially if the cultivar tends to accumulate excess copper, zinc or nickel. The necrosis of cells may also be related to burning by salts from evaporated guttation droplets. High rates of fertilizer and low soil pH may aggravate this problem.

Management

Cultural practices — Proper levels of nutrients and a soil pH of 6.0 to 6.8 should be maintained. Foliar applications of potassium chloride may help reduce this disorder. Cabbage intended for long-term storage should not be film wrapped or coated with a biopolymer. Susceptible cultivars should be stored at 3 to 4°C instead of 0 to 1°C. Controlled atmosphere storage reduces black speck.

Resistant cultivars — Cultivars resistant to black speck are available.

Selected references
Cox, E.F. 1977. Pepper spot in white cabbage - a literature review. *ADAS Q. Rev.* 25:81-86.

(Original by L.S. Bérard)

▶ 8.29 Gray speck *Fig. 8.29*

Gray speck may be present on cabbage at harvest, but the symptoms develop mainly during early storage. This disorder may be easily confused with black speck.

Symptoms The grayish discoloration of epidermal cells near the base and on the convex surface of outer head- leaves is typical of this disorder. The gray discoloration is due to the thickness of the waxy bloom. The cells actually become brown from phenol deposition on the cell wall. Lesions may be 1 to 3 mm in diameter and scattered, or they may grow together to form irregular patches of variable size in the interveinal areas but mostly along the main and larger lateral veins *(8.29).* Lesions may expand to include stomatal cells, but they do not originate with stomata. The lesions are not sunken as in black speck.

Causal agent Heavy soils, soils with a low pH, and high nitrate levels all have been implicated in this disorder. Low levels of manganese and high levels of zinc are associated with affected tissues of susceptible cultivars.

Management

Cultural practices — Growers should avoid excessive levels of soil nitrate during growth and harvest cabbage after a few fall frosts have occurred. Maintaining the soil pH around 6.5, providing adequate levels of soil organic matter, and following a crop rotation program helps to produce heads with good keeping quality. Controlled atmosphere storage reduces gray speck on many cabbage cultivars, but not with the cv. April Green.

Resistant cultivars — The cultivars Polinius, Houston Evergreen, Hidena, Slawdena, and Green Winter have good storage potential.

Selected references
Bérard, L.S., B. Vigier, R. Crête and M. Chiang. 1985. Cultivar susceptibility and storage control of grey speck disease and vein streaking, two disorders of winter cabbage. *Can. J. Plant Pathol.* 7:67-73.

(Original by L.S. Bérard)

▶ 8.30 Necrotic spot *Fig. 8.30*

Necrotic spot is a minor storage disorder of cabbage.

Symptoms Two types of necrotic spot are recognized. Type I appears as uniformly spaced, dark lesions, 1 to 5 mm in diameter, on the leaves or midribs *(8.30).* The lesions are ill-defined initially, but become sharp and sunken as the epidermis and parenchyma cells collapse. Type II appears as spots or cavities of similar size in the pith of the main stem of the head. The lesions of necrotic spot are larger than those of black speck, but smaller than those of black spot and are not confined to the top leaves as in black spot.

Causal agent The cause of necrotic spot is unknown. The disorder is variable among cultivars and seasons.

Management

Cultural practices — Controlled atmosphere storage tends to promote necrotic spot on the cultivars Superslaw, Danish Ballhead, Quick-Green Storage and Hitoma.

Resistant cultivars — Resistant cultivars are available.

Selected references
Bérard, L.S., B. Vigier and M.A. Dubuc-Lebreux. 1986. Effects of cultivar and controlled atmosphere storage on the incidence of black speck and necrotic spot in winter cabbage. *Phytoprotection* 67:63-73.

(Original by L.S. Bérard)

▶ 8.31 Vein streaking *Fig. 8.31*

Vein streaking is a storage disorder of cabbage that develops early in the storage period. Symptoms vary with the season. This disorder causes only minor losses, but trace levels of the disorder can be found in most years.

Symptoms Vein streaking is characterized by superficial brown or black markings on the epidermis of the midrib at the base of the concave surface of the outer leaves *(8.31),* with occasional extension on lateral veins. At present, vein streaking is considered distinct from gray speck because of the differential response among cultivars, storage treatments, and the distinct sites at which symptoms occur.

Causal agent The exact cause of vein streaking is not known. The brown discoloration of cells is due to the deposition of phenols on epidermal cell walls at sites where the wax bloom is thin. High levels of nitrate increase symptom severity.

Management

Cultural practices — Nitrate fertilizer should not be applied in excess. Controlled atmosphere storage usually reduces vein streaking in most cultivars, but not every year.

Resistant cultivars — No cultivar is fully resistant. The least susceptible cultivars do not keep well in storage.

Selected references (see gray speck, 8.29)
Bérard, L.S., M.A. Dubuc-Lebreux and J. Vieth. 1987. Étude histologique de la bigarrure nervale, de la griselure du limbe et de la médiane noire, trois désordres du chou en entrepôt. *Can. J. Plant Sei.* 67:321-329.

(Original by L.S. Bérard)

▶ 8.32 Frost-induced disorders *Figs. 8.32a-e*

Black blotching
Black spot
Epidermal detachment
Frost blemishing
Redheart

Cabbage heads intended for storage are usually harvested in late fall and may be exposed to frost. Although cabbage tissues freeze at –0.8°C, they can tolerate a few cycles of freezing and thawing if not subjected to mechanical shock. However, cooling and thawing rates and severity of freezing temperatures may affect tissue structures and metabolism resulting in disorders that may appear in the field or later in storage.

Black blotching

This is a superficial leaf disorder that develops during storage, possibly due to early fall frosts. Individual spots develop and may grow together to form blotchy areas. The spots are circular, 1 to 3 mm in diameter, with pin-point dark centers and a grayish or brown halo delimited by a dark line *(8.32a).* Spots are usually found on the convex surface of the leaf or on the midrib near or below the equator of the head.

Black spot

This disorder appears as large, black, interveinal necrotic areas, 1 to 5 cm in diameter, located on the top leaf of the cabbage head after several months of storage *(8.32b).*

Epidermal detachment

This disorder may result from repetitive superfreezing and thawing of the exposed outer- head leaves. The epidermis turns white and becomes detached from the parenchyma, creating a blistering effect *(8.32c).* This symptom usually appears on the epidermis of veins on the concave surface of the outermost head leaf. With severe frosts, epidermal detachment may occur on the epidermis of veins on the convex surface, in interveinal areas of both surfaces, and on leaves deeper in the head. Symptoms highly conspicuous in the field may disappear during storage as leaves begin to wilt.

Frost blemishing

This disorder is characterized by large, white, circular to triangular areas (*8.32d)* on the exposed top-head leaves and occurs after severe frosts in the field.

Redheart

This disorder is caused by freezing for more than 24 hours in the field or in storage. The damage is often irreversible. After thawing, leaf tissues several layers deep appear glassy. Outer tissues often retain their healthy appearance. Loose heads may become watery and collapse. A fetid odor may be detected in storage rooms. The internal tissues of affected heads become tan or reddish, and later may dry to a papery texture (*8.32e*). A darkened zone usually delimits affected areas from healthy areas. Heads exposed to severe frost lose their dormancy and senesce more quickly. Symptoms similar to redheart may occur in controlled atmosphere storage, if cabbage is exposed to abnormally low levels of oxygen or high levels of carbon dioxide.

Management ***Cultural practices*** — Cultivars that mature within the growing season should be selected and late planting should be avoided. If possible, the crop should be harvested before periods of severe frost. If the crop has been exposed to frost, the heads should be allowed to thaw completely before being harvested, and mechanical shock should be avoided. Growers should monitor for glassiness of inner head-leaves at harvest. For long-term storage, especially in controlled atmosphere, growers should select lots that have not been exposed to frost. Cabbage that has superficial frost injury and shows symptoms of epidermal detachment at harvest should not be placed into long-term storage. Outer head- leaves affected by black blotching or black spot can be trimmed after storage. Storage rooms should be ventilated and maintained at constant temperature slightly above 0°C (controlled atmosphere storage is 3°C) and high humidity.

Selected references
Isenberg, F.M.R. 1979. Controlled atmosphere storage of vegetables. *Hortic. Rev.* 1:337-395.

(Original by L.S. Bérard)

▶ 8.33 Other storage disorders

Dormancy
Ethylene
Maturity

Dormancy

At harvest, the apical bud within the head is dormant. During the storage period, dormancy is gradually lost and the lateral buds and apical meristem within the head begin to grow. The outer leaves of the head lose their metabolic reserves and begin to wilt, become yellow or brown, and develop a papery texture. Outer leaves may also abscise from the stem. These are symptoms of general senescence and usually occur late in storage. At that time, the head also becomes highly susceptible to rot.

Ethylene

Severe leaf-yellowing and abscission of leaves deep within the head are symptoms of ethylene exposure in early storage. Ethylene concentrations as low as 1 ppm are sufficient to speed natural senescence of stored cabbage heads. Cabbage should not be stored with fruits, such as apples, that produce ethylene. The atmosphere within a storage must be renewed periodically. In controlled atmosphere storage, levels of oxygen and carbon dioxide should be maintained at 3 and 5%, respectively, to inhibit the effects of ethylene.

Maturity

Winter cabbage intended for long periods of storage must be harvested at the proper stage of maturity. Heads that are immature at harvest usually remain green in storage and often become flaccid because they easily lose water. Overmature heads can be recognized because they are white or yellow-green and their outer leaves may be reddish or bleached from frost injury (see frost-induced disorders, 8.32). Transverse cracking of veins, leaf abscission and splitting of heads, either naturally or by mechanical shock, are also signs of overmaturity.

(Original by L.S. Bérard)

NEMATODE PESTS

▶ 8.34 Northern root-knot nematode *Fig. 6.20*

Meloidogyne hapla Chitwood

Symptoms Crucifers usually show less damage than other vegetables and are considered tolerant or resistant. Very small, spherical galls on roots may be difficult to recognize. With heavy infestations, maturity may be delayed and yield reduced. For a complete description and management strategies, see Carrot, 6.20; see also Management of nematode pests, 3.12.

▶ 8.35 Root-lesion nematode *Fig. 16.38T1*

Pratylenchus penetrans (Cobb) Filip. & Stek.

Symptoms include wilting and stunting in patches in heavy infestations; leaves become yellow. Secondary roots become necrotic, with dried areas. For a complete description, see Potato, 16.38; see also Management of nematode pests, 3.12.

▶ 8.36 Stubby-root nematodes

Paratrichodorus allii (Jensen) Siddiqi
Paratrichodorus pachydermus (Seinhorst) Siddiqi
Paratrichodorus spp.
Trichodorus spp.

This group of nematodes is not well established in Canada and has caused only minor damage to a few gardens in southern Alberta.

Symptoms Affected plants become stunted and chlorotic. Roots proliferate abnormally but appear not to grow in length and their extremities may be somewhat swollen. For a complete description, see Potato, 16.39; see also Management of nematode pests, 3.12.

▶ 8.37 Sugarbeet cyst nematode *Figs. 5.14a,b*

Heterodera schachtii Schmidt

This nematode attacks most cruciferous crops, including broccoli, Brussels sprouts, cabbage, cauliflower, kale, kohlrabi, radish, rutabaga, and turnip.

Symptoms Typically damage is most noticeable in patches where nematode densities are high. Infected plants are stunted and outer leaves wilt, yellow prematurely and die. Heart leaves are more numerous than normal but reduced in size. Tap roots are short and stunted, and lateral root development is excessive, giving a whiskered appearance to the tap root. In summer, pin-head sized, white or brown cysts can be seen on washed roots, particularly in the root axils. For a complete description, see Beet, 5.14; see also Management of nematode pests, 3.12.

INSECT PESTS

▶ 8.38 Alfalfa looper *Fig. 8.38*

Autographa californica (Speyer)

The alfalfa looper occurs in western Canada, with sporadic outbreaks in southern Alberta and British Columbia, where this species can be more important than the cabbage looper on cruciferous crops.

The larvae chew ragged holes in the leaves of most vegetable crops, sometimes defoliating them.

Identification This moth (family Noctuidae) lays eggs that are yellow. Like the cabbage looper, the larva *(8.38)* has three pairs of narrow, wavy, white lines on the back and a broad, white lateral line. In contrast, the alfalfa looper's lateral line extends almost to the lower margin of the spiracles, the head is brownish green and has a black line through the eyes; there are no legs on abdominal segments three and four in either species (8.38, *8.40c).*

Life history Two or more generations per year are usual in southern British Columbia. Pupae overwinter among crop residue. Adults emerge early in the spring and are active at night. They fly long distances and local populations may be augmented by migrants from the south. Eggs are laid directly on host plants.

Management

Monitoring — Pheromone traps are used to monitor alfalfa looper moths but action thresholds for chemical control have not been established in Canada.

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(Original by H.S. Gerber and J.A. Garland)

▶ 8.39 Aphids *Figs. 8.39a,b; 16.4la,b*

Cabbage aphid *Brevicoryne brassicae* (L.)
Green peach aphid *Myzus persicae* (Sulzer)
Turnip aphid *Lipaphis erysimi* (Kaltenbach)
Turnip root aphid *Pemphigus populitransversus* (Riley)

The green peach aphid (see Potato, 16.41) and the turnip aphid are widespread in Canada. The cabbage aphid, which is usually the most injurious of the crucifer-infesting aphids, is transcontinental in Canada. The turnip root aphid occurs but is not often recorded as a pest in Canada.

Aphids are most abundant on crucifers during dry weather. Their eggs are laid in the fall and overwinter on woody plants. Summer generations feed on cabbage, rutabaga, other crucifers and other vegetable crops. Aphid populations vary greatly on different cruciferous crops.

Damage On crucifers, all above-ground parts of the plants are attacked, including the flower-head. High populations of these aphids cause leaves to wither and plants to be stunted. Aphids may also transmit turnip mosaic virus. On rutabaga crops in southwestern Ontario, the green peach aphid is the most important vector of turnip mosaic virus but many aphid species can transmit this virus.

Identification The way the aphid colony is aggregated on the host plant is useful for field recognition: the green peach aphid is more uniformly distributed, whereas the cabbage aphid usually has a closely clumped distribution *(8.39a,b).*

The wingless form of the cabbage aphid has a dusky, gray-green abdomen with dark bands. It is covered with a mealy, gray-white wax. Its head lacks well-developed antennal tubercles and is nearly flat. Its antennae and other appendages are dark but paler at the base of each segment.

Life history Depending on the time of year and the nutritional quality of host plants, sexual forms of aphids may develop, mate, and lay eggs. Otherwise, only females are present, giving rise to live young without mating (parthenogenesis). Winged forms develop on the overwintering hosts and move to summer hosts in late spring, becoming most abundant in early summer, especially in dry weather. Aphids generally decline in early to mid-September because of the development of winged forms, a general decline in reproductive activity, an increase in the length of time to reach maturity, a decrease in the nutritional quality of the host plant, and an increase in the abundance of biocontrol agents.

Management strategies

Monitoring — No definite thresholds have been developed for crucifer-feeding aphids in Canada but, after head formation, the threshold is near zero on broccoli, cauliflower, cabbage and particularly Brussels sprouts because aphids are a serious contaminant. Prior to the development of the marketable portion of the crop, relatively high populations can be tolerated. However, because infective aphids inject a toxin, even a few aphids per plant may be serious. On rutabaga, low levels of the green peach aphid, as few as 8 to 10 per leaf, can increase partheno- genetically during warm, dry weather and quickly and completely colonize the upper third of the plant.

Biological control — Naturally occurring predators, parasites and pathogens are often effective later in the growing season. Damaging populations of aphids may appear early in the season before biocontrol agents become abundant.

Chemical control — Foliar sprays may be necessary to prevent serious crop loss and rejection of shipment, particularly when chemical control against other cruciferous pests has destroyed predators and parasites while having only a limited effect upon the aphids.

Control of the aphid vectors of turnip mosaic virus on rutabaga is not easy because they have such a diverse range of hosts and chemical insecticides are ineffective for practical control of migrant, winged aphids. Weekly applications of a light oil are highly effective in delaying and reducing aphid-transmitted turnip mosaic virus infection. The oil interferes with the acquisition and transmission of virus by feeding aphids. For proper application, growers should use 1100 L per hectare of a 1 to *2%* solution. High pressure mist nozzles are needed for effective application; drop nozzles improve coverage of leaf undersurfaces. Oil sprays should be applied weekly during periods of aphid activity until the roots have sized, which is generally late in August. To minimize phytotoxicity, oil should not be applied in bright sunlight or in combination with other spray materials, and chemical insecticides should not be applied within 24 hours of an oil treatment.

Chemical insecticides for aphid control are not needed unless early populations of the cabbage aphid and weather conditions warrant treatment. If aphid populations are sufficiently high to cause wilting, leaf-curl or stunting of plant growth, then it is usually too late to avoid damage by spraying.

(Original by D.T. Lowery, D.G.R. McLeod and L.W. Stobbs)

▶ 8.40 Cabbage looper *Figs. 8.40a-f; 3.7x-z*

Trichoplusia ni (Hübner)

The cabbage looper is an important pest of cruciferous crops in Ontario. It may be less important in other, southern areas of eastern and central Canada and British Columbia. It is not a problem on cruciferous crops in Newfoundland. In British Columbia, it is surpassed in importance by the alfalfa looper (see 8.38).

The cabbage looper does not overwinter in large numbers in Canada. Most infestations start from moth invasions from the south in July and August. In most areas, one generation is usual per season but three generations can develop in warmer areas of southwestern Ontario.

The primary cruciferous hosts are broccoli, Brussels sprouts, cabbage and cauliflower. Other hosts are beet, celery, lettuce, parsley, pea, potato, spinach, tomato and garden flowers, such as carnation, nasturtium and mignonette.

Damage The cabbage looper is generally a minor pest in the more northern regions of Canada. In the southern regions, control of the insect is essential for production of marketable cruciferous vegetables during years of heavy moth invasion. One cabbage looper larva can eat 65 cm^2 of leaf tissue during its development; most damage is done by the last two larval instars. Major concerns are damage to the marketable portion of the crop *(8.40a),* particularly the underside of cabbage and cauliflower heads and the flowers of broccoli, and the presence of larvae in the marketed crop.

The impact of the cabbage looper varies with the region, the crop, and the proposed use of the crop. This insect is not known to disseminate plant pathogens but larval damage to plants may allow entry of secondary organisms.

Identification The egg of this moth (family Noctuidae) is round and pearly white *(8.40b).* The larva is light green with three pairs of wavy white lines on the back and a lateral, white or pale yellow line that is only slightly wider than the dorsal and subdorsal lines *(8.40c).* The head is green without lateral marks. The legs on abdominal segments three and four are vestigial

(8.40d). The cabbage looper first-instar larva has a black head and part of the thorax is black. At rest or when disturbed, the larva raises the middle of its body into a loop. This is a characteristic posture. Mature larvae are 35 to 40 mm in length. The pupa, in a loose cocoon, is light green initially and darkens as it matures *(8.40e).* The adult *(8.40f)* is mottled gray-brown with a wingspan of about 38 mm. It has a silver-white mark on each forewing and, when newly emerged, there is a tuft of raised scales that resembles a collar on the thorax.

Life history Eggs are laid singly or in groups of two or three near the edge of leaf undersides. The larvae usually hatch in three to four days. They feed on the leaf undersides on cabbage and cauliflower heads, and in the flowers of broccoli, developing through five instars in two to three weeks. Pupae are encased in a loose cocoon, which usually is attached to the underside of a leaf. The pupal stage lasts about two weeks. Moths are most active late in the evening.

Management

Monitoring — Populations of adults of the cabbage looper may be monitored by pheromone traps to indicate peak flight-periods. The severity of infestations of larvae on crop plants can be estimated when larvae of the imported cabbageworm and the diamondback moth are monitored. Procedures for monitoring the imported cabbageworm may be used for the cabbage looper. However, because cabbage looper larvae are more difficult to kill with chemical insecticides than larvae of the imported cabbageworm, direct observation to estimate numbers of larvae present on crop plants is superior to indirect monitoring methods, such as estimating the proportion of plants infested or the number of fresh feeding-sites.

The economic threshold varies depending on the crop and the use that is to be made of it. For example, on crops destined for processing, there is a near-zero tolerance for larval contamination, which is the major concern. No head damage to early season cabbage grown for fresh market is tolerated. Some damage to late-season cabbage can be tolerated because the head is trimmed more extensively. The tolerance for damage to the head and inner wrapper leaves of cauliflower and broccoli is minimal, as is the tolerance for damage to crops marketed for their foliage, such as kale.

Biological control — Several parasitic wasps and flies attack larvae of the cabbage looper. The eggs and young larvae are preyed upon by various ants, beetles, bugs and spiders, but viruses are the most valuable biocontrol agents. The cabbage looper larvae are susceptible to infection by several viruses, which cause high mortality of field populations, particularly late in the season. A nuclear polyhedrosis virus is the most common. In some locations, as in southern Ontario, nearly all larvae on plants in late August may be infected by this virus *(3.7x).* It is an effective control agent but has not yet been developed as a commercial insecticide.

The bacterial insecticide *Bacillus thuringiensis* Berliner is the preferred treatment for cabbage looper control (*3.7y,z*), particularly near the time of harvest when pesticide residues are a concern. It is not toxic to or infective in mammals and has no impact on non-target organisms.

Chemical control — The first- to third-instar larvae of the cabbage looper may be controlled by chemical insecticides at concentrations similar to those usually recommended for the imported cabbageworm and the diamondback moth. Higher dosages are required to control later-instar larvae. The first application usually is required in late June on early crops, and in mid-July on late-season crops in areas where the cabbage looper is more of a problem.

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(Original by J.G. Stewart and R.P. Jaques)

▶ 8.41 Cabbage maggot *Figs. 8.41 a-g*

Delia radicum (L.)

The cabbage maggot occurs throughout Canada and is an important pest of all cruciferous crops. It has also been reported on beet, celery, and onion but these are probably erroneous identifications. Other hosts include wild mustard and wild radish.

Damage Cabbage maggot larvae generally feed on the roots of host plants. When numerous, they will destroy or severely stunt the development of young plants. Infestations on larger plants can retard growth, reduce yield, and lower quality. During cool, moist weather, survival of eggs and newly emerged larvae is highest, and root damage is usually most severe.

First-generation maggots, which are the progeny of flies from overwintered pupae, usually cause the most severe damage because the early season weather favors egg and larval survival. Field-seeded stem crucifers, and crops transplanted after mid-June usually escape severe damage because they are well established when the summer genera tions of maggots appear, fewer cabbage maggot eggs survive in areas of summer drought, and egg and larval predators are more abundant and active in July and August.

In late summer or during prolonged dry weather, oviposition and larval development may occur in above-ground stems. Emerging larvae tunnel into the stem tissue at the base of the leaves, which necessitates removal of the affected leaves prior to marketing. Chinese cabbage is especially prone to damage from maggots arising from eggs laid at the base of leaves and often suffers the loss of several leaves because of this type of feeding *(8.41b).*

The cabbage maggot is a problem in production of radish, rutabaga and summer turnip throughout the growing season because the maggot attacks the marketable part of the plant. Wounds caused by first-generation larvae result in scar tissue that persists as unsightly rough areas, reducing the market value of the crop. Larvae of subsequent generations produce furrows *(8.41a,c)* on or near the surface of the roots. These furrows do not heal prior to harvest and, if not removed by trimming, can render the crop unmarketable. Also, larvae in the roots at harvest tunnel inward, resulting in serious storage and marketing problems. The cabbage maggot is particularly important on radish because the presence of even a few maggots may render the crop unsaleable.

Production of marketable rutabaga or summer turnip depends upon adequate control of damage by the cabbage maggot. The effect of the maggot on cabbage, cauliflower, broccoli, Brussels sprouts and kale may be less severe, but infestations may reduce the size of the plants and the quality and quantity of the marketable product, or in extreme cases cause the plants to wilt and die.

Identification Cabbage maggot (family Anthomyiidae) eggs are similar in appearance to those of the seedcorn maggot. However, eggs of the cabbage maggot may be seen with the aid of a hand magnifier to have longitudinal striations and a groove that extends along their ventral aspect. In contrast, the egg of the seedcorn maggot has net-like surface sculpture and the ventral groove extends only about a third of the length of the egg. The legless larvae and pupae (puparia) of the two species can be distinguished at the ventral posterior of the body; the cabbage maggot has a pair of median tubercles that are forked at their apex, whereas the tubercles of the seedcorn maggot are not forked.

Life history In Canada, the cabbage maggot overwinters as pupae *(8.41f)* and may complete two or three generations a year, depending on the weather and soil conditions. The onset of fly emergence in the spring and the period over which flies emerge vary with climate. Therefore, the number of generations of cabbage maggot that attack a specific crop depends on the time of planting and the time of harvest of the crop in relation to the climate. Eggs are laid in the soil near cruciferous plants *(8.41d).* The larvae *(8.41e)* feed on fine root hairs of the plant and eventually burrow into the taproot below ground-level.

In southwestern Quebec, flies *(8.41g)* that emerge from overwintered pupae begin to lay eggs in the middle of May, and oviposition continues until the end of June. Maximum oviposition usually occurs in the first week of June. The larvae may complete development in less than three weeks. Adults of this generation appear in early July and usually lay fewer eggs than the parent flies. Larval survival is also lower during the warmer, drier weather of summer. Subsequent summer-generations overlap, resulting in continuous egg laying until the end of October. The life cycle in southern Ontario and southwestern British Columbia is similar, but the larvae of summer generations do little damage in southern Ontario owing to high temperatures and dry soil conditions during the summer months. The maggot can cause damage throughout most of the growing season in cooler areas.

In eastern Ontario, flies from overwintered pupae begin to lay eggs after the accumulation of 200 degree-days above 4.4°C, measured from March 1, which closely approximates full bloom of serviceberry *(Amelanchier* spp.), and peak egg laying corresponds to full bloom of McIntosh and Cortland apples. Adult emergence is later in the Atlantic provinces and in northern regions of Quebec and Ontario, because the climate is cooler. Fly emergence in the spring in the Prairie provinces coincides with the first blossoms of Saskatoon berry, *Amelanchier alnifolia* Nutt., and pin cherry, *Prunus pensylvanica* L. In many areas of Canada, eggs can be found by the last week of May, and peak activity for adults from overwintered pupae occurs between 8 to 20 June and 7 July (corresponding to the six- to nine-leaf stage of rutabaga), and continues until after mid-July. Flies of this generation begin to emerge in late July. These flies, and those from subsequent summer-generations lay eggs from early August until mid- or late September.

Management

Monitoring — Yellow-pan water-traps can be used to capture adults, thereby monitoring seasonal activity, but they are unreliable as quantitative indicators of infestation potential.

Chemical control — Protection of stem crucifers can be obtained by an application of insecticide in the planting water, as a drench after planting, or both. To protect early crops, whether field-seeded or transplanted, treatment is usually required at the time of planting. If field seeding or transplanting is done after mid-June, treatment may not be necessary because the root system is well developed when eggs are being laid, egg desiccation may occur, and destruction of both eggs and larvae by beneficial organisms is more likely. Successive and later plantings may need to be protected, and an insecticide is nearly always necessary to protect crops of radish, rutabaga and summer turnip.

Radish: An adequate level of protection can be obtained by treating the seed with an insecticide at the time of seeding, or by the application of a granular insecticide in the furrow along the row.

Rutabaga: Crops planted in May are attacked by the first two generations of the maggot. Crops planted in June are subject to attack by the second and third generations. Although plants at the four-leaf stage or earlier may be less attractive than older plants, growers should not delay treatment until damage has occurred. At least two and sometimes three drench treatments of an insecticide may be required, depending on the insecticide and climatic conditions. The first treatment is applied in the furrow below the seed at the time of seeding. One or two drench applications at five- to six-week intervals may be required, one of which should be timed to give protection during the peak of egg laying. Storage crops should receive an additional treatment applied as a soil drench with a large volume of water. The effectiveness of chemical control in preventing cabbage maggot damage to rutabaga crops in Canada is variable, and some registered insecticides at the recommended rates do not prevent damage. To minimize damage, non-storage crops should be seeded early and harvested before mid-August.

Summer turnip: A single application of an insecticide at the time of seeding is necessary; treatment consists of a granular insecticide in the furrow or a spray applied along the row.

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(Original by C. Ritchot, D.C. Read, M.Y. Steiner and D.G. Harcourt)

▶ 8.42 Diamondback moth *Figs. 8.42a-g*

Plutella xylostella (L.)

The diamondback moth occurs but apparently does not overwinter in Canada. Annual infestations arise from adults carried northward by favorable winds from winter breeding sites in the United States. These migrants arrive during early spring, often prior to the planting of cruciferous crops. The first generation in southern Canada develops largely on cruciferous weeds.

The diamondback moth attacks virtually all cultivated cruciferous crops, wild Cruciferae, and some cruciferous ornamentals. In Canada, the most important vegetable hosts are broccoli, Brussels sprouts, cabbage and cauliflower. The diamondback moth is also a serious pest of canola in western Canada.

Damage The first-instar larva *(8.42c)* mines the leaf tissues. Older larvae feed on the lower leaf-surface, chewing irregular patches in the foliage. Only the upper epidermis may remain intact on severely damaged leaves, giving the leaf a silvery appearance *(8.42a).* Older larvae feed on the florets of broccoli and cauliflower and bore into the edible portions of Brussels sprouts and cabbage. On rutabaga, larvae occasionally damage the crowns.

The diamondback moth is not known to disseminate plant pathogens but larval damage to plants may allow entry of secondary organisms.

Identification The diamondback moth (family Plutellidae; Yponomeutidae also is used) gets its name from three silvery white, diamond-shaped marks that are distinguishable when the adult is at rest with its wings folded *(8.42g).* The egg *(8.42b)* is less than 0.5 mm in length, oval, and yellowish to pale green. Larvae *(8.42c,d)* may reach 12 mm in length. They are relatively hairless, green to gray-green, and subcylindrical. They wriggle when disturbed and suspend themselves on silk threads. When mature, they pupate in a loose, open-mesh cocoon *(8.42e).* The pupa is less than 8 mm long. Initially it is pale green but it darkens as it matures. The adult *(8.42f,g)* is gray-brown with a wingspan of about 13 mm.

Life history Eggs are laid singly or in small groups, usually on the smooth, upper leaf-surfaces of the host plant. They hatch in four to six days, depending on ambient temperatures. The larvae feed on the lower leaf-surface and pass through four instars during a 10- to 14-day period. The mature larva spins a cocoon on the host plant, typically on the lower leaves but not infrequently on the wrapper leaves of cabbage or among the florets of broccoli and cauliflower. Pupation occurs within 24 hours and the adults emerge in about one week. Development from egg to adult averages 25 days in July and August in southern Ontario. The adults become active at dusk and mate within 24 hours of emergence. A female lays an average of 160 eggs during a lifespan of about two weeks; its fecundity is related to the protein content of the plant on which its larva fed. The thermal requirement for a generation is 283 degree-days above 7.3°C. There may be as many as three to six generations per year.

Management This insect is regarded as an occasional pest of cruciferous crops in most areas of Canada but an infestation may build from endemic to epidemic levels from one generation to the next. Producers must constantly be alert for sudden population eruptions. Control usually is obtained by the same treatments as applied for aphids and other butterfly and moth larvae. However, Brussels sprouts are particularly prone to feeding damage and may require special control measures.

Monitoring — Pheromone and sticky traps can be used to monitor moth flights. The procedures for monitoring the larvae are as described for the imported cabbageworm. Monitoring should begin in early summer.

Cultural practices — After harvesting early season cruciferous crops, such as rape greens and early transplanted cabbage, particularly in warm, dry weather, any remaining foliage should be disked into the soil. Sprinkler irrigation helps to discourage development of this pest while enhancing crop growth.

Biological control — The diamondback moth is attacked by several species of parasitic wasps. In southern Ontario, the most important is *Diadegma insulare* (Cress.), which does not overwinter in association with its host and has limited impact during the host's first two generations. However, during the third and subsequent generations, *D. insulare,* together with *Microplitis plutellae* Muesbeck and *Diadromus subtilicornis* (Grav.), gradually overtake the host. The bacterial insecticide *Bacillus thuringiensis* Berliner provides control in Canada, but resistance has evolved elsewhere in field populations of the diamond back moth.

Chemical control — Foliar applications of short-resid- ual-life chemical insecticides should be applied as necessary. Resistance to carbaryl and permethrin was documented in Nova Scotia and Prince Edward Island in 1990.

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(Original by J.G. Stewart and D.G. Harcourt)

▶ 8.43 European earwig *Figs. 8.43a-d*

Forficula auricularia L.

The European earwig occurs throughout Canada; it is abundant in eastern and central Canada and in southern British Columbia but is not common in the Prairie provinces.

Vegetable hosts include cabbage and other crucifers, celery, lettuce, sweet corn, and Swiss chard. The earwig eats plants and insects, switching readily from one to the other, with a preference for lichens and mosses. The presence of earwigs in fresh produce and contamination from their frass may sometimes be a problem. The impact of earwigs on commercial crop production in Canada appears to be negligible.

Damage Young nymphs may feed on seedlings. Older nymphs and adults are more likely to eat holes in leaves and chew into cabbage heads *(8.43a).*

Identification The European earwig is characterized by a pair of unsegmented, forceps-like appendages (cerci) at the anal end of the abdomen. In the female, the cerci curve inward at the tips; in the male, they are more curved and longer than in the female *(8.43b).* The adult, which usually arches the cerci over its back when disturbed, is dark red-brown. Its wings are short. Nymphs are pale brown, and their wings and cerci are much reduced or absent *(8.43d),* depending on their age.

Life history The European earwig overwinters as an adult, usually in pairs in a nest in the soil. Eggs *(8.43c)* are laid in late winter and the male is driven from the nest, leaving the female to brood the eggs. The eggs hatch during May, although the time of hatching varies with the region. A second clutch of eggs hatches about the end of June, and there may be a third clutch of eggs. Immature earwigs *(8.43d)* molt four times. During the first two instars, the young nymphs generally stay with the female, foraging at night and returning to the nest during the day. At this stage, the nymphs are most subject to mortality from excessive moisture and fungal diseases. Later, they forage more widely and shelter on the soil surface, maturing by late August. The European earwig tends to remain very localized. Adults disperse by crawling or by flight, but their chief means of spread is through man-assisted transport in soil, on equipment, or with plant material. Their habit of sheltering in any available hiding place readily enables them to be transported to new areas.

Management The European earwig seldom causes significant damage to commercial vegetable crops, although sporadic damage to individual fields and home gardens in some areas may warrant control.

Cultural practices — A practical trap can be constructed out of boards with grooves cut in them. Two boards are placed together on the ground with the grooves aligned to form a crawl space. For most effectiveness, the traps should be serviced daily and the earwigs killed. Trapping for young nymphs is done before they disperse from the nest. This same method also is effective against adults.

Biological control — The few known parasites and diseases of earwigs include the fly *Triarthria setipennis* (Fallén) (syn. *Bigonicheto spinipennis* (Meigen)), the nematode *Mermis nigrescens* Dujardin, and a poorly known fungal pathogen described in 1889 as *Entomophthora forficulae* Giard. The fly is established in British Columbia and Newfoundland. No biocontrol agents are commercially available.

Chemical control — Recommendations for chemical control of earwigs on vegetable crops have not been developed in Canada. In most cases, the European earwig is probably controlled by chemicals used against other pests. Insecticidal baits are available commercially for use in gardens and around buildings. Baits commonly consist of fish oil in bran combined with an insecticide.

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(Original by L.M. Crozier)

▶ 8.44 Flea beetles *Figs. 8.44a-e; 10.14a,b*

Cabbage flea beetle *Phyllotreta albionica* (LeConte)
Crucifer flea beetle *Phyllotreta cruciferae* (Goeze)
Garden flea beetle *Phyllotreta robusta* LeConte
Hop flea beetle *Psylliodes punctulata* Melsheimer
Horseradish flea beetle *Phyllotreta armoraciae* (Koch)
Striped flea beetle *Phyllotreta striolata* (Fabricius)

These flea beetles are largely specific to crucifers, feeding in Canada on broccoli, Brussels sprouts, cabbage, Chinese cabbage, horseradish, kale, kohlrabi, radish, rutabaga and summer turnip. They also feed on canola, mustard and cruciferous weeds.

The cabbage flea beetle is native to North America. It occurs from British Columbia to Manitoba.

The crucifer flea beetle (*8.44a; 10.14a*) was introduced on the west coast of North America from Europe in the early 1920s. It was abundant in the Prairie provinces by the late 1930s and early 1940s, and it spread eastward to reach Ontario by 1954 and Quebec and New Brunswick soon afterward. It has become the dominant flea beetle in fields of canola, particularly in the most southerly parts of the canola-growing area, and it is the predominant cruciferfeeding flea beetle across much of southern Canada.

The hop flea beetle is native and present in low numbers across most of Canada. It feeds on a wide variety of crucifers and other crops, such as garden beet, rhubarb and hop (see Herbs and Spices, 10.14).

The striped flea beetle *(8.44b)* was introduced to North America, probably before 1800. By the early 1900s, it was prevalent from the Atlantic provinces to British Columbia. It has long been considered the most common and regularly occurring of the vegetable flea beetles. In Saskatchewan, it is only abundant along the northern fringe of agriculture. Adults of this flea beetle have little feeding preference but show a marked oviposition preference for the plant on which they developed.

Other crucifer-feeding flea beetles, such as the garden flea beetle and the horseradish flea beetle, are incidental and sporadic pests in Canada. The horseradish flea beetle feeds chiefly on horseradish (see Herbs and Spices, 10.14).

Damage During outbreaks, peak numbers of 800 to 1200 flea beetles per m^2 are common. Most damage to crucifers occurs when overwintered adults feed on the cotyledons *(8.44a)* and the first true leaves of young plants early in the spring. By chewing at the stem below ground, they may cause severe, post-emergence seedling losses. Direct-seeded crops are especially vulnerable. Feeding results in small, round holes on the cotyledons and small leaves, giving the plant a “shot-hole” appearance *(8.44d; 10.14b).* A heavy infestation may destroy a young crop and necessitate reseeding, especially in hot, dry weather. Damage may be worse on light, sandy soils.

Cruciferous transplants may suffer less than direct-seeded plants but small, tender transplants that are newly set out can be killed by extensive feeding during warm, dry weather. Extensive feeding by the adult beetles *(8.44c)* also reduces yield because of reduced plant vigor and delayed, uneven maturity. When the plants have reached the six- to eight-leaf stage and are taller than 15 cm, only severe defoliation affects head weight and quality *(8.44e).* By then, plants are well established and have a greater ability to compensate for loss in leaf area.

Feeding by flea beetle larvae on the roots of radish and rutabaga may affect the appearance and marketability of those crops, whereas root damage is not a serious problem in cole crops. On radish and rutabaga crops, injury by flea beetle larvae may be masked by the presence of the cabbage maggot.

Warm, dry conditions accelerate flea beetle development and the appearance of the new generation of adults. There is a potential for severe crop damage by the newly emerged adults, and for severe crop losses the following year. A long, cold spring and/or high rainfall in May or June tend to reduce the severity of damage and economic loss.

Crucifer flea beetles have not been considered serious as pests in British Columbia or Atlantic Canada, but they are a major limitation to successful production of cruciferous vegetables in Ontario and the Prairie provinces, where control of the insects is necessary over large areas in most years. In Alberta, flea beetles have been implicated in stand reduction and yield losses in cabbage and, by inference, other cole crops. In Quebec, flea beetles on cruciferous crops are more sporadic and, although occasional damage occurs, the use of foliar sprays there is not routine.

There is no proof of disease transmission in crucifers by flea beetles in Canada, but research in New York State indicates that the crucifer flea beetle has a limited potential as a vector of the bacterium that causes black rot. Species of *Phyllotreta* and *Psylliodes* are known to spread turnip mosaic virus in Europe.

Identification Flea beetle (family Chrysomelidae) adults (*8.44a; 10.14a)* are small, 2 to 3 mm long, with shiny dark forewings (elytra) and enlarged hindleg segments (femora). Some species have yellow stripes on the elytra *(8.44b).* The adults jump when disturbed. The larvae must be reared to the adult stage for identification to species, which may require consultation with a specialist.

Life history The crucifer-feeding flea beetles are well adapted to the climate in Canada. Winter mortality is usually low, but there are times when their numbers may be severely reduced after several successive severe winters. In Canada, all crucifer-feeding flea beetles are considered to have a similar life cycle with one generation per year. The adults overwinter in leaf litter or occasionally in soil, and they may be found along fencerows, windbreaks and headlands around fields or, less frequently, within cultivated fields. Emergence from overwintering sites begins with the first extended period of warm weather in spring, peaking about mid-May. Adults feed on cruciferous weeds and volunteer canola, moving onto cruciferous crops when those emerge. Eggs are laid in the soil near the roots of host plants, or sometimes on the roots, from the end of May until early July.

The larvae feed on the roots of the host plants. The prepupal and pupal stages develop in the soil in an earthen cell, and the adults emerge from late July onward. The beetles feed on whatever crucifers are present and seek hibernation sites in late September and early October. Development from egg to adult may take as little as seven weeks, making a second generation possible in some years.

Weather plays an important role in flea beetle activity and the extent of crop damage. Adults are very active in hot, windy weather in both spring and autumn. Flight occurs above 20°C, and the adults invade cultivated fields. They feed most actively when conditions are sunny, warm and dry. Cool, damp weather reduces adult activity and the intensity of their feeding, and during inclement weather they shelter in cracks and crevices in the soil. They prefer to attack plants and foliage exposed to bright sunlight, such as seedlings, isolated plants, or plants in widely spaced rows. Shade seems to inhibit their activity.

Management

Monitoring — Infestations may be patchy or sporadic, but most fields are threatened annually in areas with a large acreage of cruciferous vegetable crops. An average of 75 flea beetles per plant can result in severe damage to a mature cabbage crop. However, because of the rapid movements of the beetles and their habit of jumping off plants at the least disturbance, it is difficult to make accurate counts. The best method of damage assessment is to look for the shot-hole injury typical of adult feeding on transplants or on cotyledons as seedlings emerge.

Cultural practices — Flea beetle damage is minimized by late or delayed seeding and by the use of high seeding rates for direct-seeded crops. Cruciferous weeds and volunteer crucifers should be controlled before emergence or transplanting of the crop, and sprinkler irrigation can be applied under warm, dry conditions to drown adult flea beetles when they are most active. Live mulches of clover, vegetable polycultures, and companion planting with marigold generally reduce the number of crucifer-feeding flea beetles, but at the expense of increased competition and lower yields. Spun polyester or other plastic materials as covers on cabbage, radish and rutabaga crops reduce flea beetle damage and promote earlier maturity of the crop, but these covers also favor diseases and weeds.

Resistant cultivars — There are cultivar differences in susceptibility in radish. Also, Chinese cabbage with dark- colored leaves has been damaged less by the striped flea beetle than cultivars with light-colored leaves. In general, crucifer-feeding flea beetles are less damaging on vegetable cultivars with a heavy, waxy bloom.

Biological control — Crucifer-feeding flea beetles are subject to low levels of natural control by native predators, parasites and pathogens, but those that do exist are ineffective for economical control. A European wasp, *Townesilitus bicolor* (Wesmael) (syn. *Microctonus bicolor),* was liberated in Manitoba during 1978-83 but has shown no evidence of establishment.

Chemical control — Chemical control usually is implemented at the first sign of shot-hole damage in cotyledons, and broccoli and other cole heads may need to be protected from cosmetic damage if they are grown near fields of canola. Granular insecticides are used to protect direct- seeded crops, and foliar treatments usually are begun when shot-hole damage appears on the leaves. For greater effectiveness and better coverage, high-volume spraying is suggested early or late in the day when evaporation and wind are at a minimum.

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(Original by JJ. Soroka)

▶ 8.45 Imported cabbageworm *Figs. 8.45a-f*

Pieris rapae (L.)
(syn. *Artogeia rapae* (L.))

The imported cabbageworm, also known as the cabbage butterfly or cabbage white, occurs on crucifers wherever they are grown in Canada. It is a serious pest in all provinces except Newfoundland.

Cruciferous vegetable hosts include broccoli, Brussels sprouts, cabbage, cauliflower, radish, rutabaga and summer turnip. Other cruciferous and non-cruciferous hosts exist.

Damage The larvae chew holes in the leaves of the plants *(8.45a).* Once the heads have started to form, feeding by a single larva can render a cabbage or cauliflower head unmarketable. When crops of broccoli, cabbage, and cauliflower become well established, the plants can tolerate extensive larval feeding. Larval frass contaminates the edible leaves and flower-heads.

The adult of the imported cabbageworm does not transmit plant pathogens but damage by the larvae may allow entry of secondary organisms.

Identification The adult is the white butterfly (*8.45f)* (family Pieridae) familiar to gardeners. The egg *(8.45b)* is elliptical, pointed at the distal end, and flat where it touches the leaf. There are 12 lengthwise ridges *(8.45c)* on its surface. When laid, the egg is creamy white; it changes to light yellow as the embryo matures. The larva is a caterpillar 30 mm in length and pale green when fully grown, with five abdominal legs, a yellow-orange stripe along the length of the dorsal midline *(8.45d),* and faint lateral bands at the level of the spiracles. Short, white hairs give it a velvety appearance *(8.45d).* The chrysalis (pupa) is *(8.45e)* about 18 mm in length, and green to brown, depending on the substrate to which it is attached. The wings of the adult are white and reach 50 mm across, females being slightly larger than males. Males have a single black spot in the middle of the forewing. Females have two such spots *(8.45f).* The forewing in both sexes has a dark patch at the apex and black scales along the leading edge. The hindwing has a small black patch at the outer edge.

Life history In Canada, except possibly in the Prairie provinces, the imported cabbageworm overwinters as a pupa. There are three to four generations of this insect per year in southern Canada. Adults first appear in early April in southern British Columbia, in late April or early May in southwestern Ontario, and in mid- to late May in eastern Ontario, Quebec and the Maritime provinces. In southern Ontario, peak oviposition of the first generation occurs in late May to early June and the generation time varies from 24 to 61 days with an average of 31 days in July and August.

Eggs *(8.45b)* are laid singly near the midrib on the lower surface of the leaf. The young larvae *(8.45c)* hatch in four to eight days after oviposition. There are five larval instars. The first three larval instars feed on the undersurface of leaves on the outside of the plant. The larger, later-instar larvae *(8.45d)* tend to move to the center or head of the plant.

Pupae of summer generations are found on the lower leaves of the crop plant *(8.45e)* or in crop residue. Overwintering pupae occur on crop residue and in debris in fencerows or other protected locations. In summer generations, the adult emerges in 8 to 20 days. After emergence, mating and oviposition commence within 24 hours, and adults are active for most of the daylight hours unless the weather is cloudy, cool or windy. Females obtain nectar from wild flowers near cultivated fields and therefore border rows of the crop tend to accumulate more eggs per plant.

Management

Monitoring — Populations of the imported cabbageworm are usually monitored in conjunction with populations of larvae of the cabbage looper and the diamondback moth. Visual estimation of the number of larvae per plant, expressed as larval units to compensate for different feeding capacity of the larvae, is a reliable basis for action thresholds for application of control measures. In excess of 90% of the crop of cabbage or other cruciferous crop may be marketable when pesticides are applied at most effective times in relation to growth of the plant and development of pest populations. The effects of feeding by pests on plant growth and on marketable yield differ with time of attack relative to plant growth and with species of crop plant (broccoli, cabbage, cauliflower).

A proposed method for estimating the number of larval units is to enumerate new feeding-sites, rather than counting the number of holes per plant, the number of larvae of each species, or the number of infested plants. A new feeding-site is a hole that appears wet and has not yet formed a callus.

Biological control — Parasites, predators and pathogens are the principal biotic factors that determine the abundance of the imported cabbageworm in Canada. For example, the wasp *Cotesia glomerata* (L.) (syn. *Apanteles glomeratus)* is an important parasite, often infesting more than 30% of larvae. *Cotesia rubecula* (Marshall) (syn. *Apanteles rubeculus),* with a biology similar to that of *C. glomerata,* is the primary larval parasite of the imported cabbageworm in British Columbia and has become established in eastern Ontario. *Pteromalus puparum* (Fabricius), another wasp, kills significant proportions of the pupal population, particularly late in the season. Flies (family Tachinidae), the most common of which is *Phryxe vulgaris* (Fallén), are parasites of pupae of this and other butterflies and moths.

A granulosis virus can cause high mortality in populations of imported cabbageworm larvae, especially late in the season. The virus kills the larvae at all stages of development and kills the pupae by infection from the larval stage. The virus, acknowledged as the key factor regulating the imported cabbageworm in some areas, is not available commercially or registered for use in Canada.

A bacterial insecticide, *Bacillus thuringiensis* Berliner, is very effective. It is the preferred treatment at present, particularly near the time of harvest.

Chemical control — Larvae of the imported cabbageworm can be killed by foliar sprays of chemical insecticides, if coverage is adequate. For early crops, treatment should be about two weeks prior to harvest. Treatment of late crops should start around mid-July and be repeated at two-week intervals as needed. Chemicals with short residual life are preferred, especially near harvest. Alternating insecticides will delay but not limit the development of resistance.

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(Original by J.G. Stewart, R.P. Jaques and D.G. Harcourt)

▶ 8.46 Purple-backed cabbageworm *Figs. 8.46a-g*

Evergeslis pallidata (Hufnagel)

The purple-backed cabbageworm is native to Europe. It occurs in the United States and Canada but is not yet recorded in Labrador or the Yukon. As a pest, this species is more important in Atlantic Canada. The species is variable in occurrence. It is spread by transport of infested produce.

The purple-backed cabbageworm attacks all cruciferous vegetables, particularly broccoli, Brussels sprouts, cabbage, cauliflower, kale, kohlrabi, rutabaga and summer turnip. Horseradish also is susceptible. Eggs of this moth may be found on shepherd's-purse, *Capsella bursa-pastoris* (L.) Medic., and sheep sorrel, *Rumex acetosella* L., but the larvae do not feed on those plants.

Damage When larvae are prevalent on rutabaga, they may completely defoliate the crop and eat holes in the roots *(8.46a).* In other cruciferous crops, they eat large holes in the foliage *(8.46b).* Specific impact studies are not available.

Identification The first evidence of infestation by the purple-backed cabbageworm (family Pyralidae) is the presence of masses of eggs on the plant foliage *(8.46c).* The entire mass is brilliant yellow with a waxy coating. Eggs within each mass are oval and flat with a translucent margin, and average 1.1 mm in length and 0.8 mm in width. Just before hatching, eggs turn brown, then black, because of the formation of the larval head, which is visible through the transparent egg shell.

The newly hatched larva is pale, watery green, and 1.5 to 2.0 mm long with many minute, dark brown tubercles on its body, each bearing one to several long setae. The fully grown larva (*8.46d*) is 20 to 22 mm long, robust and covered with setae. It is purple-brown above and ash-gray beneath, with a conspicuous, narrow, yellow lateral band along the entire length of the body and a narrow white band bordering the lower margin of the yellow band.

The cocoon is oval, 12 to 15 mm long and 5 to 7 mm wide, and lined internally with dark gray silk. Its exterior becomes covered with soil particles attached by a viscid substance, making it resemble a lump of soil. The pupa (*8.46e)* inside the cocoon is light to dark brown.

The moth *(8.46g)* has a wingspan of 22 to 28 mm. It is straw-yellow with irregular, dark brown lines. Males and females are similar in size and color.

Life history The purple-backed cabbageworm has one generation per year in Canada. Eggs *(8.46c)* are laid in compact masses on the undersurface of the lower leaves of susceptible plants. Larvae hatch in four to eight days, feed on the leaf undersurfaces for two to three weeks, hide between leaves during the day, then wander off the host plant and spin cocoons just below the soil surface. Winter is passed as a prepupa (larva) in the cocoon *(8.46f);* pupation occurs the following June. The moth escapes through a loosely constructed end on the cocoon. Local moth dispersal is assisted by wind.

Management

Monitoring — The presence of large numbers of egg masses and/or larvae feeding extensively on leaves indicate a need for action, especially on rutabaga.

Cultural practices — Cultivation or plowing in late fall or early spring helps to bury cocoons, potentially destroying the prepupae and preventing the moths from reaching the surface. A trap crop of summer turnip can be used to attract the moth and usually becomes heavily infested when grown alongside cabbage or rutabaga. Periodic spraying of the trap crop kills the larvae and protects the adjoining crop.

Biological control — The purple-backed cabbageworm is unusually free from natural enemies. No larval diseases have been found. *Bracon montrealensis* Morrison and *Meteorus autographae* Muesebeck are parasitic wasps with potential for biocontrol.

Chemical control — The purple-backed cabbageworm is controlled by the same insecticides that are applied against other crucifer-feeding caterpillars. Foliar sprays can be used to control young larvae.

(Original by R.F. Morris)

▶ 8.47 Red turnip beetle *Figs. 8.47a-c*

Entomoscelis americana Brown

The red turnip beetle is native to the western North American interior plains between latitudes 45 and 68°N. In Canada, it is most abundant in the agricultural parkland zone of the Prairie provinces and occurs in the interior of British Columbia south of 55°N.

The red turnip beetle is an occasional pest of cruciferous crops, mostly in home gardens. The beetle feeds as a larva and adult on cruciferous plants, including canola, mustard, vegetable crops and weeds.

Damage Damage, which usually involves large numbers of beetles completely destroying the crop, is greatest in June when newly emerged adults migrate from fields where they previously infested cruciferous crops and weeds. Newly emerged beetles do not fly but may walk several hundred metres in large numbers in search of food. The arrival of hundreds of beetles can quickly devastate home- garden crucifers.

Identification Adults *(8.47a)* of the red turnip beetle (family Chrysomelidae) are large, about 10 mm long and 5 mm wide, with longitudinal, broad, red and black bands on the forewings (elytra). Eggs are brown, and about 1.5 mm in length *(8.47c).* Larvae *(8.47b)* are wrinkled and dull black, 1 to 2 mm long when just hatched, and 10 to 15 mm long when fully grown. Pupae are orange, and 6 to 10 mm in length *(8.47c).*

Life history The red turnip beetle has one generation per year. The eggs *(8.47c)* occur singly or in small clusters and overwinter on or near the soil surface, or beneath soil lumps or crop residue. Larvae hatch soon after the snow melts in spring, from late March to early May, and larval development normally is completed by the end of May. The larvae enter the soil to pupate and pupal development takes about two weeks. Adults emerge during the first three weeks of June. They briefly feed, then hibernate for about one month, reappearing in late July and August. They fly, feed, mate and oviposit until late October or the onset of cold weather, when they die.

Management

Monitoring — Fields of canola stubble or other sources of infestation close to cruciferous vegetable crops or gardens should be monitored in June when the red turnip beetle adults are easily seen.

Cultural practices — Fall or spring cultivation of infested fields will kill red turnip beetle eggs. Weed control in the spring eliminates volunteer canola and other cruciferous hosts.

Biological control — No parasites of the red turnip beetle are known and the incidence of predation and disease is very low.

Chemical control — Feeding by the red turnip beetle on cruciferous weeds is beneficial, so chemical control usually is discouraged. However, when large numbers of adult red turnip beetles are invading a cruciferous crop, insecticidal applications to the infested area are effective.

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(Original by W.J. Turnock)

▶ 8.48 Other insect pests *Figs. 16.49c-e*

Leatherjackets
White grubs

Leatherjackets are the maggot-like larvae of crane flies (family Tipulidae), of which there are many species in Canada. These larvae are fleshy, grayish black, and about 2.5 cm in length at maturity. They overwinter in the soil and feed on the roots of cruciferous seedlings and transplants. Growers can reduce overwintering leatherjacket populations in areas where they are a problem by practicing clean cultivation, which minimizes the potential for the larvae to damage roots in the spring.

(Original by J.A. Garland)

White grubs (*16.49c-e*) can damage cruciferous root crops, such as rutabaga and turnip, by chewing holes in the sides of the roots. Damage usually occurs when the crop is planted on recently broken land or in weedy fields that already are infested. Chemical control is rarely necessary. (For more information, see Potato, 16.49.)

(Original by K.P. Lim and J.C. Guppy)

OTHER PESTS

▶ 8.49 Gray garden slug *Figs. 11.27c; 18.43*

Deroceras reticulatum (Müller)

The gray garden slug (for other species of slugs, see Lettuce, 11.27) occurs in home gardens in urban areas across Canada. It attacks cruciferous and other vegetable crops, and ornamental lilies *(Convallaria* and *Lilium* spp.).

Damage Cruciferous seedlings are subject to serious injury by infestations of this slug near fences or hedgerows where dense plant growth provides shelter. Later in the season, they may damage Brussels sprouts by climbing the stalks and eating holes in the tender young sprouts; on rutabaga and summer turnip, they make holes that can be mistaken for damage by the purple-backed cabbageworm. During wet weather in the fall, this and other slug species may squeeze between the leaves of Brussels sprouts or cabbage, thereby contaminating the marketable parts.

Identification The gray garden slug (family Limacidae) is 35 to 50 mm long at maturity, and gray-white or creamy flesh-colored with irregular gray markings. The breathing pore is at the rear of center on the right side of the mantle, the body tapers abruptly posteriorly, and the slime is clear.

Life history There is one generation per year. Eggs overwinter. The slugs mature during the growing season, sheltering in tall grass or other plant growth. They breed in the fall, then die (see Lettuce, slugs and snails, 11.27).

Management

Monitoring — Slime trails and excreta are persistent and readily seen signs of slugs. Beer is a strong attractant and has been used to monitor slug populations. Monitoring should begin early, during seedling emergence and after transplanting.

Cultural practices — Vegetable crops should not be planted in low, flat, wet or recently plowed land that has been left idle for several years. Effective ways to keep slug populations low, in the short term, are clean cultivation and removal of sheltering sites along hedgerows and fences.

Biological control — The ground beetle *Calosoma frigidum* Kirby occurs across Canada. It, and birds, snakes, frogs and toads may destroy many slugs, but they are seldom present in sufficient numbers to be effective in vegetable crop fields or home gardens.

Chemical control — Several pesticides have given good to excellent control of slugs in experimental trials but have been of limited value in field operations. Compounds containing tin, aluminum, or a carbamate with sulfur are most effective. The timing of treatments is critical. Compounds for the home garden usually contain the active ingredient metaldehyde, which is also an attractant. Chemicals work best when the slugs are most active, between midnight and early morning when conditions are cool and moist, particularly during periods of dry weather.

(Original by D.C. Read)

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