INTRODUCTION

Collecting, conservation and utilization of plant genetic resources and their global distribution are essential components of international crop improvement programmes.

Inevitably, the movement of germplasm involves a risk of accidentally introducing plant quarantine pests along with the host plant material; in particular, pathogens that are often symptomless, such as viruses, pose a special risk. In order to minimize this risk, effective testing (indexing) procedures are required to ensure that distributed material is free of pests that are of quarantine concern.

The ever increasing volume of germplasm exchanged internationally, coupled with recent, rapid advances in biotechnology, has created a pressing need for crop-specific overviews of the existing knowledge in all disciplines relating to the phytosanitary safety of germplasm transfer. This has prompted FAO and IBPGR to launch a collaborative programme for the safe and expeditious movement of germplasm reflecting the complementarity of their mandates with regard to the safe movement of germplasm. FAO has a long-standing mandate to assist its member governments to strengthen their Plant Quarantine Services, while IBPGR’s mandate - *inter alia* - is to further the collecting, conservation and use of the genetic diversity of useful plants for the benefit of people throughout the world.

The aim of the joint FAO/IBPGR programme is to generate a series of crop-specific technical guidelines that provide relevant information on disease indexing and other procedures that will help to ensure phytosanitary safety when germplasm is moved internationally.

The technical guidelines are produced by meetings of panels of experts on the crop(s) concerned, who have been selected in consultation with the relevant specialized institutions and research centres. The experts contribute to the elaboration of the guidelines in their private capacities and do not represent the organizations to which they belong. FAO, IBPGR and the contributing experts cannot be held responsible for any failures resulting from the application of the present guidelines. By their nature they reflect the consensus of the crop specialists who attended the meeting, based on the best scientific knowledge available at the time of the meeting.

The technical guidelines are written in a short, direct, sometimes ‘telegraphic’ style, in order to keep the volume of the document to a minimum and to facilitate
updating. The guidelines are divided into two parts: The first part makes general recommendations on how best to move germplasm of the crop concerned and mentions available intermediate quarantine facilities when relevant. The second part covers the important pests and diseases of quarantine concern. The information given on a particular pest or disease does not pretend to be exhaustive but concentrates on those aspects that are most relevant to quarantine. In general, references are only given on the geographical distribution of the diseases and pests, their seed transmission and methods of indexing.

It should be realized that the information on pest distribution is strongly influenced by the intensity of research carried out in a given country or region and should therefore be considered as relative.

The naming of legume crops is often confusing. A lists the accepted Latin and vernacular names of major cultivated legume species is given in the Appendix.

The present guidelines were developed at a meeting held in Arnhem, the Netherlands from 16 to 22 April 1989. The meeting was convened by the Research Institute for Plant Protection (IPO) and sponsored by the Directorate General for International Cooperation (DGIS) of the Netherlands Ministry of Foreign Affairs.
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CONCEPTUAL GUIDELINES

A. Germplasm

- All legume germplasm collections should be maintained free of known seed-associated pests (seed-borne or seed-transmitted in the case of fungi and bacteria; seed-transmitted in the case of viruses). Descriptor data should be obtained from pest-free germplasm.

- Only seedlots certified to be free of such pests should be distributed.

- In recipient countries, seedlots should be established and maintained for one generation under conditions of isolation (temporal and/or spatial) or containment, with periodic inspection, testing and roguing.

B. Breeding lines

- Legume seedlots to be exchanged among breeding programmes should be produced under conditions of isolation (with appropriate chemical protection) or containment, with periodic inspection and roguing to eliminate seed-associated pests.

- Seedlots should be tested for seed-associated pests and certified by the appropriate regulatory agency before distribution.

C. Commercial seedlots

- Commercial seedlots should continue to be subject to current regulatory procedures.
TECHNICAL GUIDELINES

A. General recommendations

- Vegetative material of legume species should go through intermediate or post-entry quarantine and should be tested for absence of viruses.
- Legume seed should not be moved internationally in pods.
- Seed should be harvested at optimal time for the crop and care taken to ensure effective drying.
- Seed samples should be cleaned to eliminate all soil, plant debris, seeds of noxious weeds, and phanerogamic parasites.
- Unless specified otherwise, seeds should be surface-disinfected (with sodium hypochlorite or a similar product) before being given appropriate fungicide and insecticide treatments.
- Seedlots suspected to contain insects should be fumigated with an appropriate pesticide.
- Parcels containing seeds should be unpacked in a closed (insect-proof) area and packing material should be incinerated or autoclaved.

B. Movement of germplasm

1. Introduction of germplasm

- Introduction of new germplasm entries should satisfy local regulatory requirements.
- Each new introduction should be grown under containment or isolation.
- Plants should be observed periodically. Plants suspected to be affected with seed-associated pests should be destroyed.
- All symptomless plants should be tested for latent infections by viruses known to occur in the place of origin of the material and in the country of
maintenance (see Table 1 on pp. 50-52). Ideally this testing should be carried out at this stage or, if not possible, it should be carried out before the germplasm is distributed (see International distribution of germplasm). Infected plants should be destroyed.

- Seed should be collected from healthy plants only.

2. Further multiplication of new introductions or rejuvenation of germplasm accessions

- Seed should be sown under containment or isolation with appropriate chemical protection.

- Plants should be observed periodically. Plants affected by seed-associated pests should be removed and destroyed.

- Seed should be collected from healthy plants only.

3. International distribution of germplasm

- Germplasm accessions that have been introduced and multiplied according to the procedures described above can be certified and distributed internationally.

- Germplasm accessions which are not yet in a pest-free state should be handled according to the same procedures as described for new introductions.

- Movement of germplasm should comply with regulatory requirements of the importing country.

- In addition to the phytosanitary certificate, a ‘germplasm health statement’, indicating which tests have been performed to assess the health status of the material, should accompany the germplasm accession.

C. Movement of breeding material

- Seeds used for the multiplication of breeding material should be pest-free.

- Breeding material under multiplication should be grown under containment or isolation with appropriate chemical protection.
• Plants should be inspected soon after emergence and periodically thereafter. Plants infected with seed-associated pests should be destroyed. For field grown plants, suitable precautions should be taken to prevent soil spread from infected plants and introduction of possible seed-associated pests from local sources of infection.

• Seeds should be harvested only from symptomless plants.

• Seed samples of appropriate size should be tested for seed-associated pests.

• When non-destructive seed health tests are available, all seeds should be tested accordingly.

• Movement of germplasm should comply with regulatory requirements of the importing country.

• In addition to the phytosanitary certificate, a germplasm health statement, indicating which tests have been performed to assess the health status of the material, should accompany the breeding material.
PESTS OF QUARANTINE IMPORTANCE

Viral diseases

1. Alfalfa mosaic virus
Alfalfa mosaic virus group; four classes of bacilliform particles c. 18 nm wide x 57,43, 35, and 30 nm long; readily transmitted in sap (Jaspars & Bos, 1980).

Host range
Occurs often symptomlessly in many legumes. Natural host range is very wide and includes over 150 species in 22 families of dicotyledons.

Symptoms
Mosaic and mottle symptoms in lucerne, but often masked at higher temperature. In soybean, brilliant yellow mottle or mosaic (calico); in common bean, cowpea and mungbean, systemic yellow mosaic. Lethal systemic necrosis may occur in pea, and wilting in chickpea. Red and white clover often exhibit mosaic.

Transmission
Readily transmitted by aphids (at least 14 species) in the non-persistent manner. Seed transmission depends on host genotype and virus strain and amounts up to c. 50% in lucerne (Beczner & Manninger, 1975; also in pollen to embryos on virus-free mother plants, but not to these plants when pollinated with infected pollen: Hemmati & McLean, 1977). Seed transmission of berseem mosaic virus (most probably alfalfa mosaic virus) in *Trifolium alexandrinum* was 60 - 70% (Mishra *et al.*, 1980). May be transmitted by hay-cutting machinery.

Geographical distribution
Worldwide.

Indexing
Mechanical inoculation of *Phaseolus vulgaris* (usually necrotic lesions); *Chenopodium amaranticolor* and *C. quinoa* (chlorotic or necrotic lesions, sometimes systemic); and *Vigna unguiculata* (necrotic lesions for certain isolates and strains). Can also be indexed using ELISA; infected seedlots can be screened by ELISA, but testing of whole seeds may also reveal antigen in coats of seeds of which the embryo is free of virus (Pesic & Hiruki, 1986).
References

2. Bean common mosaic virus
Potyvirus group; flexuous filamentous particles c. 750 nm; low to medium concentration in systemically-infected plants; readily transmitted in sap; comprises highly different strains (Drijfhout, 1978; Morales & Bos, 1988).

Host range
Mainly found in *Phaseolus* species, mungbean (Kaiser et al., 1968) and some wild legumes such as *Rhynchosia minima*. Also reported from *Lupinus luteus* (Frencel & Pospieszny, 1979). Several other legumes including cowpea are suspected but unconfirmed hosts. Non-leguminous artificial hosts include *Nicotiana benthamiana* and *N. clevelandii*.

Symptoms
Vein-banding mosaic of dark green areas along main leaf veins, sometimes accompanied by leaf malformation (curling or blisters). Mosaic-resistant bean genotypes may show local and/or systemic necrosis (Drijfhout, 1978).

Transmission
Transmitted in a non-persistent manner by several aphid species, mainly *Aphis fabae* and *Myzus persicae*. Transmission via seed of common bean may be high, depending upon bean cultivar and virus strain (Morales & Castano, 1987). Seed transmission also reported for mungbean (*Vigna radiata*) (up to 25%: Kaiser et al., 1968), phasemy bean (*Macroptilium lathyroides*) (Provvidenti & Braverman, 1976), tepary bean (*Phaseolus acutifolius*) (7-22%: Provvidenti & Cobb, 1975) and urdbean (*Vigna mungo*) (2-10%: Agarwal et al., 1979).

Geographical distribution
Worldwide.
Indexing
Highly susceptible common bean genotypes, such as Dubbele Witte, show both mosaic and leaf distortion. Bean cvs Topcrop and Widusa develop local and systemic necrosis when inoculated with necrosis-inducing strains of the virus. ELISA.

References


3. Beanpod mottle virus
Comovirus group; isometric particles c. 30 nm; high concentration in plants; readily transmitted in sap (Semancik, 1972).

Host range
Common bean and soybean. Also reported from Desmodium paniculatum (Moore & Walters, 1969).

Symptoms
Plant stunting, severe leaf mosaic and pod mottle in common bean. Leaf mottle and puckering and pod and seed-coat mottling in soybean.

Transmission
By Cerotoma trifurcata and other leaf beetles. Seed transmission in soybean reported only once (0.1%; Lin & Hill, 1983).
**Geographical distribution**
USA.

**Indexing**
Serology.

**References**

**4. Bean yellow mosaic virus**
Potyvirus group; flexuous particles c. 750 nm; transmitted in sap (Bos, 1970); various strains exist such as the bean mosaic, pea yellow mosaic and pea necrosis strains (Bos et al., 1974).

**Host range**
Many legumes, including common bean, faba bean, pea, chickpea, cowpea, *Crotalaria spectabilis*, soybean and perennial legumes and some non-legumes such as squash, spinach, *Freesia, Gladiolus* and a number of bulb crops (Derks et al., 1980).

**Symptoms**
Causes mosaics and necrosis in legumes depending upon host genotype and virus strain.

**Transmission**
By many aphid species in the non-persistent manner and via seed in some legume species such as faba bean (Quantz, 1954; 0.1-2.4%: Kaiser, 1973; 0.1-0.2%: Fiedorow, 1980), pea (Dickson, 1922), white sweet clover, and white and yellow lupin (3-6%: Zschau, 1962; 6.2%: Corbett, 1958).

**Geographical distribution**
Worldwide.

**Indexing**
Diagnostic hosts are selected cultivars of common bean, faba bean and pea (‘Perfection’ type peas are immune), and *Chenopodium amaranticolor* and *C. quinoa*. ELISA and immuno-specific electron microscopy are sensitive tests for detection and recognition.
References

5. Blackeye cowpea mosaic virus
Potyvirus group; flexuous, filamentous particles, c. 750 nm; moderate concentration in cowpea plants; readily transmitted in sap (Purcifull & Gonsalves, 1985). The virus is closely related to cowpea aphid-borne mosaic virus (Purcifull & Gonsalves, 1985; Dijkstra et al., 1987), from which it differs in host range and serology (Taiwo et al., 1982) but perhaps not sufficiently to treat the latter as a distinct virus (Dijkstra et al., 1987).

Host range
Occurs naturally in cowpea (Anderson, 1955; Lima et al., 1979), asparagus bean (V. unguiculata var. sesquipedalis) (Tsuchizaki et al., 1984), common bean, mungbean (Green, 1985), soybean (deviant strain, Dijkstra et al., 1987), and Crotalaria spectabilis (Anderson, 1955). Experimentally transmissible to various other leguminous crop species and several test plants of a number of families.

Symptoms
Prominent mosaic, mottle, green vein-banding and distortion in susceptible genotypes. When occurring together with cucumber mosaic virus, severe stunting in cowpea (Pio-Ribeiro et al., 1978) and rugose mosaic in asparagus bean (Chang, 1983).
Transmission
By *Aphis craccivora*, *Macrosiphum euphorbiae* and *Myzus persicae* in a non-persistent manner (Anderson, 1955), and probably by many other aphid species. Transmitted up to 30.9% in seed of several cowpea genotypes (Anderson, 1957; Zettler and Evans, 1972), and in mungbean (0.6-2.5% in 7 out of 13 lines tested with a virus closely related to the virus and adzuki been mosaic virus (Green, 1985).

Geographical distribution
Possibly wherever cowpea is grown.

Indexing
Serologically, in agar (SDS, pyrrolidine), but more reliably by ELISA.

References
6. **Blackgram mottle virus**
Possibly carmovirus group, isometric particles c. 28 nm; transmissible in sap (Scott & Hoy, 1981).

**Host range**
Blackgram (urd) (*Vigna mungo*) in seeds of which it was first detected (Phatak, 1974).

**Symptoms**
Mottling and stunting in blackgram.

**Transmission**
Transmitted in sap, by beetles (*Cerotoma trifurcata* and *Epilachna varivestis*), and via seed of blackgram (8%; Phatak, 1974).

**Geographical distribution**
Australasia, India, Thailand.

**Indexing**
On assay hosts (*Cyamopsis tetragonoloba*, *Macrotyloma uniflorum*, *Phaseolus lunatus*, *P. vulgaris* ‘Pinto’, ‘Puregold’). Latex serology (the virus is a good immunogen), ISEM.

**References**

7. **Broad bean mottle virus**
Bromovirus group: isometric particles c. 27 nm; high concentration in plants; readily transmitted in sap (Gibbs, 1972).

**Host range**
Only found in faba bean, but infectious to 12 of 27 legumes (including chickpea, lentil and pea, which suffered severely, and soybean, *Phaseolus vulgaris*, *Trifolium* spp. and *Melilotus albus*) and 9 non-legumes.

**Symptoms**
Faba-bean plants react with mottling, marbling or diffuse mosaic often associated with leaf malformation and sometimes with plant stunting and bushy growth. Some genotypes may show necrosis,
Transmission
Artificially by beetles (Acalymma trivittata, Diabrotica undecimpunctata and Colaspis flavida) and possibly weevils (Sitona lineatus). Via seed of faba bean when occurring together with bean yellow mosaic virus (Murant et al., 1974; Makkouk et al., 1988).

Geographical distribution
North Africa, Portugal, Sudan, Syria, UK.

Indexing
Test plants (Chenopodium amaranticolor, C. quinoa, cotyledons of Cucumis sativus), ELISA.

References

8. Broad bean stain virus
Comovirus group; angular isometric particles, c. 28 nm; high concentration in plants; transmissible in sap (Gibbs & Smith, 1970). Pea green mosaic virus and pea seed-borne symptomless virus are strains (Musil et al., 1983).

Host range
Only found in faba bean (Vicia faba), lentil, pea, vetch and hybrid clover (Makkouk et al., 1986,1987; Musil et al., 1983; Tapio, 1970) but infectious to chickpea, some cultivars of Phaseolus bean and mostly symptomlessly to a number of wild Leguminosae. Not infectious to non-legumes (Makkouk et al., 1987).

Symptoms
Mild mottling in faba bean and diffuse mottling in pea. No symptoms in most other artificial hosts. Seeds of infected faba bean may show a characteristic necrotic pattern of the testa around the periphery of the seed.

Transmission
By weevils (Apion vorax and Sitona spp.). Via seed of faba bean: up to 10% (Gibbs & Smith, 1970) or 2.7% (Jones, 1978), even when unstained (Makkouk et al., 1987); and in seeds of pea (Kowalska & Beczner, 1980), lentil (Makkouk & Azzam, 1986), and Vicia palaestinae, a symptomless artificial host of the virus (Makkouk et al., 1987).
**Geographical distribution**
Europe, North Africa, Sudan and West Asia (Makkouk et al., 1987). Detected in Australia and in experimental plots in China, but probably eradicated.

**Indexing**
In leaves, ground seeds and developing embryos of faba bean with ELISA. Virus sometimes detectable in cotyledons while not in embryonal axis (Makkouk et al., 1987).

**References**

**9. Broad bean true mosaic virus**
Comovirus group; angular isometric particles c. 28 nm; high concentration in plants; transmitted in sap (Gibbs & Paul, 1970).

**Host range**
Only found in faba bean (Gibbs & Paul, 1970) and pea. Artificially transmissible to several legumes but not to non-legumes (Gibbs & Paul, 1970).
**Symptoms**
Malforming leaf mottle and mosaic, often masked at high temperature. Cyclical development of disease (Paul & Quantz, 1959).

**Transmission**
By weevils (*Apion vorax* and *Sitona* spp.) and via seed of faba bean (up to 17%: Blaszczak, 1970, 1974; Cockbain *et al.*, 1976; Jones, 1978, 1980).

**Geographical distribution**
Europe and northwest Africa (Gibbs & Paul, 1970), and China (Ji, 1987). Found in South Australia in crops grown from imported seed, but no evidence of spread (Boswell & Gibbs, 1983).

**Indexing**
Diagnostic hosts are faba bean and pea, with *C. amaranticolor* and *N. clevelandii* as insusceptible hosts; ELISA.

**References**
10. **Cowpea aphid-borne mosaic virus**

Potyvirus group; flexuous, filamentous particles, c 750 nm; moderate concentration in plants; readily transmitted in sap (Bock & Conti, 1974). The virus is closely related to, if not identical with blackeye cowpea mosaic virus (Dijkstra et al., 1987); and probably also azuki bean mosaic virus, occurring in *Vigna angularis* in Japan (Hino, 1962).

**Host range**

Occurs in cowpea. Experimentally transmissible to various other leguminous crop species and various test plants of the Chenopodiaceae, Cucurbitaceae, Lamiaceae and Solanaceae.

**Symptoms**

Severe mosaic, mottle and distortion in susceptible genotypes. All degrees of susceptibility exist. A range of types (strains), widely differing in symptomatology in cowpea, have been identified (Bock, 1973; Purcifull & Gonsalves, 1985; Rossel and Thottappilly, unpublished).

**Transmission**

By various aphid species (Lovisolo & Conti, 1966; Bock, 1973) and at variable rates in seed of several cowpea genotypes (up to 40%; Kaiser & Mossahebi, 1975; Aboul Ata et al., 1982). Azuki bean mosaic virus was also found to be seed-transmitted (Tsuchizaki et al., 1970a; 1970b).

**Geographical distribution**

Possibly wherever cowpea is grown.

**Indexing**

Serologically, in agar (SDS) but more reliably by ELISA. Various, biologically and/ or serologically distinct strains identified.

**References**


bean and from soybean, and the relationships between blackeye cowpea mosaic virus and cowpea aphid-borne mosaic virus. *Neth. J. Plant Path.* **93**:115-133.


11. **Cowpea mild mottle virus**

Affiliation uncertain; formerly grouped under the Carlavirus; filamentous, rather rigid particles, c. 650 nm; high concentration in plants; readily transmitted in sap (Brunt & Kenten, 1974).

**Most range**


**Symptoms**

Mild mosaic, mottle in soybean and a few susceptible cowpea genotypes. Symptoms in soybean are generally mild. Prominent chlorosis, stunt and rugose symptoms in common bean. Certain strains cause bright yellow mosaic in soybean (Rossel and Thottappilly, unpublished).

**Transmission**

Transmitted by whiteflies (*Bemisia tabaci*). Seed transmission reported (up to nearly 100%) for cowpea, soybean and common bean (Brunt & Kenten, 1973) and for soybean (0.5%: Thouvenel *et al.*, 1982). Seed transmission in soybean could not be confirmed in
Nigeria (Rossel and Thottappilly, in preparation). Similar studies in India have shown low (0.5-2%) seed-borne infection rates (Reddy, in preparation). Virus also detected in mungbean seed obtained from Tanzania (Mink, pers. comm.)

**Geographical distribution**
Probably worldwide in the tropics. Common in leguminous crop and weed species in Africa.

**Indexing**
Serologically by ELISA.

**References**

**12. Cowpea mosaic virus**
Comovirus group; isometric particles, c. 25 nm; high concentration in plants; readily transmitted in sap (Van Kammen & De Jager, 1978). This virus was originally described as cowpea yellow mosaic virus (Chant, 1959; Swaans & van Kammen, 1973).

**Host range**
Occurs in cowpea (Chant, 1959; Bock, 1971), also reported from groundnut and soybean in Japan, from *Crotalaria juncea* (Ladipo, 1988) and *Cajanus cajan* (Bock, 1971), sporadically found in soybean in Africa (Rossel and Thottappilly, unpublished). Experimentally transmissible to other leguminous crop species, and some test plants like *Chenopodium* spp. and *Nicotiana benthamiana.*
Symptoms
Severe mosaic, mottle and distortion in susceptible genotypes. All degrees of susceptibility exist. Numerous cowpea genotypes have high levels of resistance (including hypersensitivity).

Transmission
By the chrysomelid beetles, *Ootheca mutabilis* and *Paraluperodes quaternus*, and by *Nematocerus acerbus* (Curculionidae) (Chant, 1959; Bock, 1971; Whitney & Gilmer, 1974). Other chrysomelid beetles also incriminated as vectors, and vectors may remain infective for 1-2 to more than 8 days (Van Kammen & De Jager, 1978). Suspected seed transmission (1-5%: Gilmer et al., 1973) could not be confirmed (Thottappilly and Rossel, 1987).

Geographical distribution
Occurs in the humid savanna and forest zones of West Africa. Also reported from some countries in East Africa: Kenya (Bock, 1971), Tanzania (Patel and Kuwite, 1982) and in Suriname, Cuba and the USA.

Indexing
Serologically, in agar or by ELISA.

References
13. **Cow-pea mottle virus**
Possibly carmovirus group; spherical particles c. 27 nm; high concentration in plants; readily transmitted in sap (Boswell & Gibbs, 1983).

**Host range**
Occurs in cowpea and bambara groundnut (*Vigna (=Voandzeia) subterranea*) (Robertson, 1966; Rossel, 1977; Shoyinka *et al.*, 1978). Experimentally transmissible to other leguminous crop species and some test plants like *Chenopodium* spp.

**Symptoms**
Severe mosaic, mottle and distortion in susceptible genotypes. All degrees of susceptibility exist. Cowpea genotypes identified which possess high levels of resistance (Allen, 1980).

**Transmission**
By the chrysomelid beetle, *Ootheca mutabilis*. Seed transmission in all three cowpea cultivars tested (up to 10%: Shoyinka *et al.*, 1978; Allen *et al.*, 1982), in inoculated plants of common bean (Shoyinka *et al.*, 1978) and in bambara groundnut (Robertson, 1966).

**Geographical distribution**
Occurs throughout the humid savanna and forest zones of West Africa.

**Indexing**
Serologically in agar or by ELISA.

**References**
14. **Cowpea ringspot virus**
Cucumovirus group: spherical particles c. 25-30 nm; low to medium concentration in cowpea; readily transmitted in sap (Phatak *et al*., 1976).

**Host range**
Found naturally in cowpeas. Also found in lima bean (*Phaseolus lunatus*) and winged bean (*Psophocarpus tetragonolobus*) (Rossel, unpublished). Experimentally transmissible to other leguminous crop species and some non-legume species such as *Chenopodium* spp., *Nicotiana glutinosa* and *N. benthamiana*.

**Symptoms**
Generally very mild and consisting of characteristic patchy chlorosis or mottle.

**Transmission**
Naturally by numerous aphid species in the non-persistent manner and through seed of cowpea (10-30%: Phatak, 1974; Phatak *et al*., 1976).

**Geographical distribution**
Probably occurs wherever cowpeas are grown.

**Indexing**
By mechanical transmission to *N. glutinosa* and serologically by agar-gel double diffusion or ELISA.

**References**

15. **Cowpea severe mosaic virus**
Comovirus group; isometric particles, c. 25 nm; high concentration in plants; readily transmitted in sap (Swaans & van Kammen, 1973; De Jager, 1979).

**Host range**
Occurs naturally in cowpea (Dale, 1949; Van Hoof, 1963; Agrawal, 1964); also found in common bean and other leguminous crops (Dale, 1949; Lin *et al*., 1982). Sporadically found in soybean (Thongmeearkom &Goodman, 1976). Experimentally transmissible only to other leguminous species.
Symptoms
Severe mosaic, mottle and distortion in susceptible genotypes. All degrees of susceptibility exist. Resistance not commonly found among cowpea germplasm.

Transmission
By several leaf-feeding chrysomelid beetles, mainly Cerotoma ruficornis and C. trifurcata. Reportedly transmitted in seed of cowpea (up to 10%: Shepherd, 1964; Haque & Persad, 1975) and of asparagus bean (Vigna sesquipedalis) (8%: Dale, 1949).

Geographical distribution
Occurs in cowpea and common bean in Latin America and the southern USA.

Indexing
Serologically in agar or by ELISA.

References
16. Cryptic (or temperate) viruses

Cryptovirus group; spherical particles c. 30 nm in diameter with segmented dsRNA of about $4 \times 10^6$; good immunogens but no mutual serological relationships (Boccardo et al., 1983; Natsuaki et al., 1986). The group includes: alfalfa cryptic virus (Boccardo et al., 1983), hop trefoil cryptic virus (Boccardo et al., 1983), red clover cryptic virus (Boccardo et al., 1983), Vicia cryptic virus (Kenten et al., 1980; Abou-Elnasr et al., 1985) and white clover cryptic virus (Boccardo et al., 1983).

Host range
Single plant species.

Symptoms
None. Not known to be of any economic importance.

Transmission
Not mechanically or by grafting. No known vector. In high rates via seed (Boccardo et al., 1983) but most probably not of quarantine importance.

Geographical distribution
Europe and Japan, probably worldwide. Rather common in cultivated legumes (Boccardo et al., 1983).

Indexing
Only after purification or by immunosorbent electron microscopy (Boccardo et al., 1983). No routine test available.

References
17. **Cucumber mosaic virus**
Cucumovirus group; spherical particles c. 29 nm; concentration variable in plants; readily transmitted in sap (Francki et al., 1979). The seed-transmitted cowpea banding mosaic virus (Prakash & Joshi, 1980) is probably a legume strain of cucumber mosaic virus.

**Host range**
Found naturally in many angiosperms, especially Cucurbitaceae and Solanaceae. Also reported from many Leguminosae such as azuki bean, chickpea, cowpea, faba bean, groundnut, lentil, lucerne, lupins, *Phaseolus* bean, *Pisum sativum* and various clovers (Bos & Maat, 1974). Legume isolates are often weakly pathogenic to non-legumes (Bos & Maat, 1974).

**Symptoms**
Symptoms vary from none to mottling and mosaic on systemicleaves, sometimes with stunting and leaf malformation. In cowpea, severe stunting and in asparagus bean, rugose mosaic when in complex with blackeye cowpea mosaic virus (Pio Riberio et al., 1978; Chang, 1983). In *Phaseolus* bean symptoms often confused with those of bean common mosaic virus (Bos & Maat, 1974; Meiners et al., 1977). Necrosis in some species, such as yellow lupin. Plants often recover.

**Transmission**
Naturally by numerous aphid species in the non-persistent manner. Artificially by mechanical inoculation. Through seed of common bean (Bos & Maat, 1974; Meiners et al., 1977), cowpea (Green, 1985), groundnut (Xu & Barnett, 1984), mung bean (Phatak, 1974; Purivirojkul et al., 1978; Iwaki, 1978), yellow and blue lupin (Golebniak, 1979; Jones, 1988).

**Geographical distribution**
Worldwide.

**Detection**
Test plants *Chenopodium amaranticolor, C. quinoa, Cucumis sativus, Vigna unguiculata; ELISA.*

**References**


18. Guar symptomless virus
Potyvirus group; flexuous particles c. 760 nm; transmitted in sap (Hansen & Lese-mann, 1978).

Host range
Cyamopsis tetragonoloba.

Symptoms
None or mild green mottle. Plants recover.

Transmission
Non-persistently by aphids and via seed (up to 70% in commercial seed: Behncken, 1983).

Geographical distribution
Found in seed from several continents. Occurs in Australia, India, Pakistan, USA.
**Indexing**

Diagnostic hosts are *Chenopodium amaranticolor*, *C. quinoa*, *Glycine soja*, *Macroptilium lathyroides*, *Macrotyloma uniflorum*, *Phaseolus vulgaris* ‘Bountiful’.

**References**


**19. Lucerne Australian latent virus**

Nepovirus group; spherical particles c. 24-27 nm with angular profiles; low concentration in plants; transmitted by mechanical inoculation (Jones & Forster, 1980).

**Host range**

Found in nature only in *Medicago sativa* and *Trifolium repens*. Experimental hosts include *Cajanus cajan*, *Cicer arietinum*, *Lupinus* spp., *Phaseolus vulgaris*, *Pisum* spp., *Trifolium* spp. and *Vigna unguiculata*.

**Symptoms**

Most susceptible host species were infected systemically without symptoms. White clover may display chlorotic line patterns seasonally.

**Transmission**

The virus spreads in nature in lucerne fields, but the mechanism is unknown. Seed transmission up to 8% in lucerne and to 9% in seed from inoculated *Chenopodium quinoa* plants (Blackstock, 1978). Pollen transmission to seed and progeny seedlings occurred in *C. quinoa* (Blackstock, 1978).

**Geographical distribution**

Recorded only from Australia and New Zealand.

**Indexing**

Diagnostic species are *Chenopodium amaranticolor*, *C. quinoa*, *Gomphrena globosa* and *Pisum sativum*. Antisera react well in gel-diffusion tests. Isolates from lucerne and white clover and their homologous antisera showed little or no cross reaction in DAS-ELISA (Forster & Morris-Krsinich, 1985).

**References**


### 20. Lucerne transient streak virus

Sobemovirus group; spherical particles c. 27-28 nm with angular profiles; low concentration in plants; transmitted by mechanical inoculation (Forster & Jones, 1980).

**Host range**

Found in nature only in *Medicago sativa*. Experimental hosts infected systemically included *Trifolium incarnatum* plus several species of *Lupinus* and *Medicago*.

**Symptoms**

Systemic vein clearing and chlorotic vein banding. Reduced dry matter yield of lucerne by 18% (Blackstock, 1978).

**Transmission**

Increasing incidence of infection with age of lucerne stands suggested that field spread occurred but the mechanism is unknown (Blackstock, 1978). All seedlings (> 200) grown from seed collected from infected plants were symptomless, but the distribution of infected plants in lucerne fields suggested that the virus could be seed-borne and it was detected serologically in 2.5% of seedlings of *Melilotus albus* (Paliwal, 1983).

**Geographical distribution**

Recorded from Australia, Canada and New Zealand.

**Indexing**

Diagnostic host species are *Chenopodium amaranticolor*, *C. quinoa*, *Medicago scutellata*, *Pisum sativum* and *Nicotiana clevelandii*. The virus is weakly immunogenic but an antiserum readily detected it in gel-diffusion tests.

**References**


21. Pea early-browning virus

Tobavirus group; straight tubular particles of two predominant lengths c. 105 and 215 x 21 nm; transmissible in sap (Harrison, 1973); broad bean yellow band virus (Russo *et al.*, 1982) is a serotype (Robinson & Harrison, 1985).

**Host range**


**Symptoms**

In pea irregular leaf, stem and pod necrosis; entire shoots may be killed; in some cultivars leaf mottling (*Bos & van der Want*, 1963). In common bean irregular leaf and stem necrosis with severe plant stunting (*Gerhardson & Ryden*, 1979; *Bos & Huijberts*, unpublished data). In faba bean infection is often symptomless (*Fiedorow*, 1980; Lockhart & Fischer, 1976), but plants may die prematurely if simultaneously infected by bean leafroll virus (*Coskbain et al.*, 1983); yellow vein banding is caused by the broad bean yellow band serotype (*Russo et al.*, 1982).

**Transmission**

In crops the disease occurs in patches and transmission is by trichodorid nematodes (*Trichodorus* spp.). Above-ground spread is by seed. Rate of transmission in pea is 1-2% (Harrison, 1973) or up to 37% (*Bos & van der Want*, 1963) and up to 10% in faba bean (*Fiedorow*, 1983).
Geographical distribution
Europe (Harrison, 1973; Kowalska, 1979; Fiedorow, 1983) and Morocco (Lockhart & Fischer, 1976).

Indexing
Inoculated leaves of Chenopodium amaranticolor, cucumber (cotyledons and foliage leaves, even when detached in petri dishes), and of common bean (primary leaves) react with characteristic local lesions in 3 - 4 days (Bos & van der Want, 1963). ELISA for detection in seeds (Van Vuurde & Maat, 1985).

References
22. Pea seed-borne mosaic virus

Potyvirus group; filamentous rods c. 12 x 770 nm; moderate concentration in plants; readily transmitted in sap (Hampton & Mink, 1975; Khetarpal & Maury, 1987).

Host range

Occurs naturally in *Lens esculenta*, *Pisum sativum*, *Vicia faba* and *V. villosa*. A few non-legume species infected experimentally.

Symptoms

Stunting, systemic vein clearing, leaf rolling, rosetting, flower distortion or abortion, small pods. Leafrolling is easily mistaken for physiological disorder. Some pea genotypes react with necrosis and premature plant death. In Yugoslavia a latent pea strain was described (Milicic & Plavsic, 1978). A lentil strain was non-pathogenic to most pea genotypes (Hampton, 1982), whereas another isolate was much more severe on peas and two other pathotypes differed on pea genotypes (Alconero et al., 1986).

Transmission

Naturally by aphids in the non-persistent manner. Artificially by mechanical inoculation. Seed-transmitted in pea (Mink et al., 1969; Alconero & Hoch, 1989) up to 95% depending on cultivar (Cockbain, 1988), in lentil up to 44% (Hampton & Muehlbauer, 1977), and in faba bean up to 3% (Musil, 1980). Infected seeds are erratically distributed in pods and on plants of pea (Musil, 1980).

Geographical distribution

Asia (India, Japan, Taiwan), Australia, Europe, New Zealand, North Africa and North America.

Indexing

Test plants: Chenopodium amaranticolor, *Pisum sativum* (especially ‘Perfection’-type cultivars immune to bean yellow mosaic virus). Efficiently in seeds with ELISA in group samples of up to 100 seeds (Maury et al., 1987).

References


23. Peanut clump virus
Furovirus group: rod-shaped particles, bipartite, 245 nm and 190 x 22 nm; transmissible in sap (Thouvenel & Fauquet, 1981b).

**Host range**
Infected naturally groundnut, chillies (*Capsicum annuum*), great millet (*Sorghum arundinaceum*). Experimentally transmissible to several dicots and monocots. High concentration in *Nicotiana clevelandii, N. glutinosa, Phaseolus vulgaris* ‘Topcrop’.

**Symptoms**
Groundnut plants are severely stunted and dark green; leaflets are smaller, not deformed; young leaflets show small chlorotic rings.

**Transmission**
Soil-borne by the fungus *Polymyxa graminis*. Seed transmitted 6-14% in groundnut (up to 20% in groundnut seeds collected from diseased plants: Thouvenel & Fauquet, 1981a). Also seed transmitted in cereal crops.

**Geographical distribution**
Burkina Faso, Côte d’Ivoire, India, Niger, Senegal and South Africa.

**Indexing**
For Indian isolates: *Phaseolus vulgaris* ‘Topcrop’ produces necrotic lesions or veinal necrosis; *Canavalia ensiformis* produces necrotic or chlorotic patches or symptomless infection, depending on isolate. For West African isolates: *Chenopodium amaranticolor* produces concentric ring spots and line pattern extending along the veins. The virus occurs in several serologically distinct isolates. Five isolates have been reported for
Indian PCV and two isolates for West African PCV. Thus serology may not be useful for detection unless antisera specific to each isolate could be obtained. However, complementary DNA probes prepared for one of the Indian isolates detected all five Indian isolates and one West African isolate.

References

24. Peanut mottle virus
Potyvirus group: flexuous rod shaped particles c. 750 nm; high concentration in plants (Bock & Kuhn, 1975; Bock, 1983).

Host range
Infects naturally groundnut, wild groundnut (Arachis chacoense), common bean, cowpea, lupins (Lupinus angustifolius and L. albus), mungbean (Vigna radiata), pea, soybean, and forage legumes such as subterranean clover and arrowleaf clover (Trifolium vesiculosum). Twenty-seven legumes (among which are several important legume crops) and 4 non-legumes have been reported as experimental hosts.

Symptoms
In groundnut mild mottle on youngest leaflets; older leaflets show upward curling of edges, interveinal depression and mild mottling. Some genotypes may not show upward curling of leaf edges. Can reduce yield of pods up to 40%.

Transmission
By aphids in the non-persistent manner; Aphis craccivora appears to be the principal vector. Seed transmission frequency: 0-8.5% (Adams & Kuhn, 1977) or 20% (Bock, 1973) to less than 1% in the majority of groundnut cultivars (Bharathan et al., 1984).
Less than 1% found in one cowpea plant introduction (Demski et al., 1983a) and in *Lupinus albus* (Demski et al., 1983b). Low percentage in seeds of navy bean (*Phaseolus vulgaris*) (Behncken & McCarthy, 1973).

**Geographical distribution**
Worldwide.

**Indexing**
*Phaseolus vulgaris* ‘Topcrop’ produces reddish-brown local lesions: non-systemic. ELISA (Bharatan et al., 1984). Seeds of groundnut can be non-destructively tested in ELISA on thin slices from apical ends of seeds (in groups of 25).

**References**

**25. Peanut stripe virus**
Potyvirus group; flexuous rod-shaped particles 730-750 nm; high concentration in several hosts (Demski et al., 1984).

**Host range**
Natural hosts are groundnut, cowpea, soybean and *Dolichos lablab*. Experimentally the virus infects 15 legumes and 8 non-legumes; preferred propagation host is *Lupinus albus*. 
Symptoms
In groundnut initial symptoms are distinct stripes or blotches on young leaflets. Older leaflets show conspicuous mosaic in the form of green islands or oak-leaf patterns, and unlike the symptoms of peanut mottle, these symptoms persist in older leaflets. Can reduce yield of pods up to 50%.

Transmission
By aphids in the non-persistent manner. *Aphis craccivora* appears to be the principal vector. Under experimental conditions the virus can be seed-borne in groundnut up to c. 40% (37%: Demski et al., 1984; 43%: Ohki et al., 1989). Under field conditions seed transmission is usually from 1 to 5%.

Geographical distribution
China, India, Indonesia, Japan, Malaysia, Myanmar (Burma), North America, Philippines, Thailand and Vietnam.

Indexing
*Chenopodium amaranticolor, C. quinoa* (local lesions). The virus reacts strongly with blackeye cowpea mosaic, bean common mosaic and soybean mosaic virus antisera and is not serologically related to peanut mottle virus. ELISA with monoclonal antibodies (Culver & Sherwood, 1988). Seeds of groundnut can be non-destructively tested in ELISA using slices from apical ends of seeds (in groups of ten) (Demski & Warwick, 1986).

References


26. **Peanut stunt virus**

Cucumovirus group; spherical particles c. 30 nm; moderate concentration in plants; readily transmitted in sap (Mink, 1972).

**Host range**

Found naturally in several legume species such as groundnut, *Phaseolus* bean, many clovers which may act as important sources of infection, and in some non-leguminous plants. Also reported from faba bean, pea and soybean. Experimentally infectious to a wide range of non-leguminous plants.

**Symptoms**

Pronounced stunting of groundnut. Necrotic or chlorotic lesions on inoculated leaves of bean, followed by systemic mottling, leaf distortion, epinasty and plant stunting.

**Transmission**

Naturally by aphids in the non-persistent manner. Experimentally by mechanical inoculation. Transmitted through seed of groundnut (0.1%: Troutman *et al.*, 1967).

**Geographical distribution**

Africa, Asia, Europe, Japan and North America.

**Indexing**

Test plants *Chenopodium amaranticolor, C. quinoa, Vigna unguiculata*; ELISA.

**References**


27. **Southern bean mosaic virus**

Sobemovirus group; isometric particles c. 30 nm; concentration high in infected tissues; readily transmitted in sap (Tremaine & Hamilton, 1983).

**Host range**

Very narrow natural host range; only leguminous species are susceptible. Occurs often in common bean, cowpea, black gram, mungbean, and, to a lesser extent, in soybean. Isolates from bean rarely infect cowpea and those from cowpea rarely infect bean. The Ghana cowpea strain infects bean systemically without symptoms.
Symptoms
Mosaic and mottle, of ten associated with leaf deformation.

Transmission
Transmitted by several species of leaf beetles (Chrysomelidae) in a circulative manner. In North America, the bean and cowpea strains are transmitted by Cerotoma trifurcata and Epilachna varivestis; in Africa, the main vector is Ootheca mutabilis. Possibly transmission through contact. Seed-transmitted in cowpea (1-40%: Shepherd & Fulton, 1962; Lamptey & Hamilton, 1974; Givord, 1981; O’Hair et al., 1981) and common bean (1-30%: Jayasinghe, 1982; Morales & Castano, 1985); probably in seed coat only (McDonald & Hamilton, 1972). Seed transmission in cowpea is enhanced by simultaneous infection with cowpea chlorotic mottle virus (Kuhn & Dawson, 1973).

Geographical distribution
Warm, temperate and tropical regions of the Americas, India and Africa. May occur in other regions as a consequence of importing infected seed.

Indexing
Phaseolus vulgaris ‘Pinto’ and ‘Top Crop’ and Vigna unguiculata ‘Clay’ are useful local lesion hosts for bean and cowpea isolates, respectively. The high concentration of virus in sap allows reliable detection using serological methods (immunodiffusion and ELISA).

References
O’Hair, S.K., Miller, J.C. & Toler, R.W. 1981. Reaction of cowpea introductions to
infection with the cowpea strain of southern bean mosaic virus. *Plant Dis.* **65**:251-252.


**28. Soybean mosaic virus**

Potyvirus group; flexuous, filamentous particles, c. 750 nm; moderate concentration in soybean; readily transmitted in sap (Bos, 1972; Irwin & Schultz, 1981).

**Host range**

Occurs in soybean; recently found in *Vicia faba* in China (Xu *et al.*, 1986) and in white lupin (Vroon *et al.*, 1988). Experimentally transmissible to only a few other legume crop species and some other test species such as *Chenopodium* spp. Certain isolates are transmissible to *Nicotiana benthamiana* (Rossel, unpublished).

**Symptoms**

Generally mild, and consisting of characteristic leaf rolling, mottle and rugose symptoms. Severe mosaic and distortion with some isolates. Only a few genotypes possess high levels of resistance and, in most cases, only to a number of isolates (Cho & Goodman, 1979).

**Transmission**

By several aphid species in the non-persistent manner. High rates of seed transmission observed in soybean greatly depending upon cultivar (Bowers & Goodman, 1979; Goodman *et al.*, 1979; Goodman & Oard, 1980) and 1.2% in one experiment with white lupin (Vroon *et al.*, 1988).

**Geographical distribution**

Occurs wherever soybean is grown.

**Indexing**

Not visually, since seed-coat mottling, though stimulated by infection by the virus, is not directly correlated with the presence of the virus in particular seeds (e.g. Ross, 1970). Serologically, in agar (SDS), but more reliably by ELISA. For testing of soybean seeds in ELISA in groups of 30 or more and the avoidance of false positives due to seed-coat infection, see Maury *et al.* (1985,1987).

**References**


29. **Soybean stunt virus**

Cucumovirus group; isometric particles c. 28-30 nm; moderate concentration in plants; readily transmitted in sap (Boswell & Gibbs, 1983).

**Host range**

Found naturally only in soybean. Experimentally infectious to 14 legumes (including *Cassia tora, Cyamopsis tetragonoloba, C. occidentalis, Dolichos lablab, Lupinus chamissonis, Medicago sativa, Phaseolus angularis, P. aureus, P. lunatus, P. vulgaris, Pisum sativum, Vicia faba, Vigna sesquipedalis* and *V. sinensis*); 15 of 24 non-leguminous species were infected.

**Symptoms**

Soybean plants exhibit mottle, leaf crinkle and stunt; some varieties exhibit vein-necrosis on the leaf apex or margin and top necrosis.
**Transmission**
Naturally transmitted by aphids in the non-persistent manner. Via seeds of soybean (up to 50%: Koshimizu & Iizuka, 1963).

**Geographical distribution**
China, Indonesia, Japan, USA, USSR.

**Indexing**
Test plants (*Chenopodium amaranticolor, Nicotiana tabacum 'White Burley', Phaseolus vulgaris ‘Monroe’*). Agar-gel double diffusion, ELISA.

**References**

**30. Subterranean clover mottle virus**
Sobemovirus group; spherical particles c. 30 nm with angular profiles; high concentration in plants; readily transmitted by mechanical inoculation (Francki *et al.*, 1988).

**Host range**
Found in nature only in *Trifolium glomeratum* and *T. subterraneum*. *Medicago truncatula* was infected systemically in experimental tests. Host range studies on this virus have been very limited.

**Symptoms**
Severe stunting with leaf mottling, reddening and distortion in subterranean clover. Barrel medic plants develop a mosaic and are stunted. Dry matter production is reduced by 60-100% following infection.

**Transmission**
The virus spreads in nature and an aerial vector is implicated but it has not been identified. The virus was found serologically to be present in up to 10% of seeds of commercial seedlots of subterranean clover and in up to 3% of seedlings obtained from such seed lots (Francki *et al.*, 1988).

**Geographical distribution**
Recorded in all southern states of Australia where subterranean clover is grown. Incidence in pastures sometimes exceeds 50%. The virus may be endemic to Australia (Francki *et al.*, 1988).
Indexing

*Pisum sativum* is a local lesion host. *Medicago truncatula* and *Trifolium subterraneum* are diagnostic hosts, while *Phaseolus vulgaris*, *Vicia faba* and *Vigna sinensis* are diagnostic non-hosts. The virus is readily detected serologically in gel-diffusion tests, ELISA and dot immunobinding assay.

References


31. Sunn-hemp mosaic virus

Tobamovirus group; rod-shaped particles, 300 nm; readily sap-transmissible.

Synonyms: *Dulichos* enation mosaic virus, southern sunn-hemp mosaic virus, *Crotalaria mucronata* mosaic virus, cowpea mosaic virus (Kassanis & Varma, 1975).

Host range

Wide among legumes: cowpea, sunn hemp (*Crotalaria juncea*), *Dolichos lablab*, *Mucuna aterrima*.

Symptoms

Mosaic, blistering and malformation of leaves.

Transmission

Readily in sap and through contact. No vector. Via seed of cowpea (17.5%: Kulthe & Mali, 1979; cowpea chlorotic spot isolate 4-20%: Kassanis & Varma, 1975). In sunn-hemp little or no seed transmission (Căpoor, 1962; Nagaich & Vashisth, 1963; Căpoor et al., 1947). The serologically distinct rosette virus of *Crotalaria juncea* with similar though slightly longer particles, was reported to be transmitted in 10 - 20% of the seeds from infected plants (Verma & Awasthi, 1976,1978).

Geographical distribution

Africa, India and North America.
Indexing
Local lesion hosts are *Nicotiana glutinosa* and *N. tabacum* ‘Xanthi nc’.

References

32. Tobacco ringspot virus
Nepovirus group; isometric particles c. 28 nm (Stace-Smith, 1983).

Host range
Wide natural host range, infecting annual and perennial herbaceous and woody species. Principal legume host is soybean, although common bean is also infected (Tu, 1981). Also found in sweet clover (*Melilotus* spp.) (Henderson & Wingard, 1934), red clover (Jones & Diachun, 1976), *Cyamopsis tetragonoloba* (Orellana, 1966), *Crotalaria* (Komuro & Iwaki, 1968), *Lotus corniculatus* (Ostazeski, 1965), *Lupinus polyphyllus* (Kowalska, 1971) and *Pisum sativum* (Stubbs, 1937).

Symptoms
Young infected soybean plants exhibit severe stunting, curvature of the terminal bud, and necrosis of most buds (bud blight), depending on virus strain and cultivar (Tu, 1986). Pods may be underdeveloped or aborted. Similar symptoms occur in soybean infected with tobacco streak virus (Fagbenle and Ford, 1967; Sinclair, 1982), indicating need for correct identification of causal virus.
Transmission
Naturally transmitted by nematodes (*Xiphinema americanum*) but transmission in soybean is inefficient. *Thrips tabaci* may be a natural vector. Readily seed-transmitted (70-100%) in soybean (Athow & Bancroft, 1959; Owusu *et al.*, 1968).

Geographical distribution
The virus is endemic in soybean production areas of North America. Also reported to occur in Egypt, Turkey, India and Sri Lanka (Hamilton, 1985).

Indexing
Mechanical inoculation to *Nicotiana clevelandii, N. tabacum, Chenopodium amaranticolor* and *Vigna unguiculata*, which are useful local lesion hosts. ELISA is applicable to seed-testing (Lister, 1978) and plant assays (Moore *et al.*, 1982).

References


### 33. Tobacco streak virus

Iloravirus group; isometric particles, 27-35 nm; readily transmitted by manual inoculation (Fulton, 1985).

**Host range**

Affects soybean (Costa and Carvalho, 1961), cowpea (Kaiser *et al*., 1982) and common bean, in which it causes red node disease (Thomas & Zaumeyer, 1960; Greber, 1971). Also reported from pea (Patino & Zaumeyer, 1959) and some clovers. Causes disease in a wide range of non-legume crops.

**Symptoms**

Bud blight on soybeans in Brazil and the USA. Early infection may lead to complete yield loss. Irregular chlorotic spots on leaves which later may be dwarfed in appearance.

**Transmission**

Thrips (*Frankliniella occidentalis* and *Thrips tabaci*) have been reported as vectors. High rates of seed transmission reported for soybean (2.6-30% depending upon cultivar: Ghanekar & Schwenk, 1974; up to 90%: Kaiser *et al*., 1982), less than 1% in cowpea (Kaiser *et al*., 1982) and up to 26% in common bean (Thomas & Graham, 1951). Also transmitted in seed of *Melilotus albus* (Kaiser *et al*., 1982) and of several non-legumes.

**Geographical distribution**

Australia, Europe, Japan, North and South America (Fagbenle & Ford, 1970) and New Zealand.

**Indexing**

Serologically in agar and by ELISA.

**References**

34. Tomato aspermy virus

Cucumovirus group; spherical particles 25-30 nm; fairly concentrated in plants; readily transmitted in sap (Hollings & Stone, 1971).

Host range

Found naturally in tomato and chrysanthemum. Experimentally infects a wide range of plants.

Symptoms

Systemic mottle, mosaic, blisters and distortion on young leaves of *Phaseolus* bean. In some varieties, yellow spots along the veins.

Transmission

Naturally by aphids in the non-persistent manner. Experimentally by mechanical inoculation. Seed transmitted in beans up to 18.7% (Wang, 1982).

Geographical distribution

Reported from Australia, Europe, India, Japan, New Zealand and North America.

Indexing

Test plants (*Chenopodium amaranticolor*, *C. quinoa*, *Nicotiana glutinosa* and *Phaseolus vulgaris*); gel-diffusion serology.

References

35. **Urdbean leaf crinkle virus**

Ungrouped spherical virus, c. 25-30 nm; transmission in sap (Beniwal, 1983).

**Host range**


**Symptoms**

Leaf rugosity, crinkling and distortion.

**Transmission**

In sap and naturally by beetles (*Henosepilachna dodecastigma*). Via seed of urdbean (18%: Kolte & Nene, 1972) and in 3 out of 49 mungbean germplasm accessions (6 -15%) at Pantnagar (Beniwal *et al.*, 1980).

**Geographical distribution**

India.

**Indexing**

Assay hosts are cucumber ‘National Pickling’, *Lagenaria cylindrica*, *Vigna aconitifolia*, *V. mungo* and *V. unguiculata*.

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<td>5-44</td>
<td>37,38</td>
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<tr>
<td><em>Lupinus albus</em></td>
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<td>35</td>
<td>Europe</td>
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<td>Peanut mottle</td>
<td>0.4</td>
<td>24</td>
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<tr>
<td><em>Lupinus angustifolius</em></td>
<td>Cucumber mosaic</td>
<td>3-34</td>
<td>4,47</td>
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<td>6</td>
<td>7,21</td>
<td>Worldwide</td>
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<td>Host</td>
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<td>% transmission</td>
<td>Reference</td>
<td>Geographical distribution</td>
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<td>Cucumber mosaic</td>
<td>21</td>
<td>84</td>
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<td><em>Macroptilium lathyroides</em></td>
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<td>5-33</td>
<td>68</td>
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<td>0.2-49</td>
<td>46</td>
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<td><em>Medicago sativa</em></td>
<td>Alfalfa mosaic</td>
<td>1-30</td>
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<td>Lucerne Australian latent</td>
<td>0-8</td>
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<td><em>Medicago truncatula</em></td>
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<td>10,65</td>
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<td>Tobacco streak</td>
<td>0-3</td>
<td>51</td>
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<td><em>Phaseolus acutifolius</em> var. latifolius</td>
<td>Bean common mosaic</td>
<td>7-22</td>
<td>69</td>
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<td>60,62</td>
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<td>0-1</td>
<td>8</td>
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<tr>
<td></td>
<td>Southern bean mosaic</td>
<td>1-30</td>
<td>43,61</td>
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<td>Tomato aspermy</td>
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<td>Bean yellow mosaic</td>
<td>5</td>
<td>26</td>
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</tr>
<tr>
<td></td>
<td>Peas seedborne mosaic</td>
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<td>53</td>
<td>Worldwide</td>
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<tr>
<td></td>
<td>Pea early-browning</td>
<td>1-37</td>
<td>15,41</td>
<td>Europe, Morocco</td>
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<tr>
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<td>Pea mild mosaic</td>
<td>15</td>
<td>19</td>
<td>New Zealand</td>
</tr>
<tr>
<td><em>Trifolium subterraneum</em></td>
<td>Subterranean clover mottle</td>
<td>3</td>
<td>30</td>
<td>Australia</td>
</tr>
<tr>
<td><em>Vicia faba</em></td>
<td>Bean yellow mosaic</td>
<td>0.1-2.4</td>
<td>48,63</td>
<td>Iran, Sudan</td>
</tr>
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</table>
Table 1. Naturally seed-transmitted viruses occurring in different legumes (cont’d).

<table>
<thead>
<tr>
<th>Host</th>
<th>Virus</th>
<th>% transmission</th>
<th>Reference</th>
<th>Geographical distribution</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>Vicia faba</em></td>
<td>Broad bean mottle</td>
<td>1-2</td>
<td>58</td>
<td>North Africa, Portugal, UK, Syria, Sudan</td>
</tr>
<tr>
<td></td>
<td>Broad bean stain</td>
<td>1-10</td>
<td>32,44,45</td>
<td>Australia*, China*, Europe, North Africa, Sudan</td>
</tr>
<tr>
<td></td>
<td>Broad bean true mosaic</td>
<td>1-17</td>
<td>11,12,20,44</td>
<td>Australia*, Europe, northwest Africa</td>
</tr>
<tr>
<td></td>
<td>Pea seed borne mosaic</td>
<td>**</td>
<td>64</td>
<td>Europe</td>
</tr>
<tr>
<td></td>
<td>Pea early-browning</td>
<td>1-10</td>
<td>28,29</td>
<td>Europe</td>
</tr>
<tr>
<td><em>Vigna catjang</em></td>
<td>Sunn-hemp mosaic</td>
<td>17</td>
<td>18</td>
<td>India</td>
</tr>
<tr>
<td><em>Vigna mungo</em></td>
<td>Bean common mosaic</td>
<td>2-10</td>
<td>3</td>
<td>India</td>
</tr>
<tr>
<td></td>
<td>Blackgram mottle</td>
<td>8</td>
<td>70</td>
<td>India, Thailand</td>
</tr>
<tr>
<td></td>
<td>Urdbean leaf crinkle</td>
<td>18</td>
<td>9</td>
<td>India</td>
</tr>
<tr>
<td><em>Vigna radiata</em></td>
<td>Bean common mosaic</td>
<td>8-32</td>
<td>49</td>
<td>Iran</td>
</tr>
<tr>
<td></td>
<td>Cucumber mosaic</td>
<td>10</td>
<td>42</td>
<td>Japan</td>
</tr>
<tr>
<td><em>Vigna sesquipedalis</em></td>
<td>Cowpea severe mosaic</td>
<td>8</td>
<td>22</td>
<td>South America, southern USA</td>
</tr>
<tr>
<td><em>Vigna unguiculata</em></td>
<td>Blackeye cowpea mosaic</td>
<td>30</td>
<td>83</td>
<td>Worldwide</td>
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<tr>
<td></td>
<td>Cowpea aphid-borne mosaic</td>
<td>7-18</td>
<td>1,50</td>
<td>Worldwide</td>
</tr>
<tr>
<td></td>
<td>Cowpea mild mottle</td>
<td>90</td>
<td>17,77</td>
<td>Worldwide</td>
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<tr>
<td></td>
<td>Cowpea mosaic</td>
<td>1-5</td>
<td>33</td>
<td>Cuba, Kenya, Nigeria, Suriname, USA</td>
</tr>
<tr>
<td></td>
<td>Cowpea mottle</td>
<td>0.2-10</td>
<td>5,73</td>
<td>Nigeria</td>
</tr>
<tr>
<td></td>
<td>Cowpea ringspot</td>
<td>10-30</td>
<td>67</td>
<td>Iran</td>
</tr>
<tr>
<td></td>
<td>Cowpea severe mosaic</td>
<td>1-10</td>
<td>22,40,71</td>
<td>Americas, Puerto Rico, Trinidad</td>
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<tr>
<td></td>
<td>Cucumber mosaic</td>
<td>15-20</td>
<td>67</td>
<td>Worldwide</td>
</tr>
<tr>
<td></td>
<td>Peanut mottle</td>
<td>&lt;1</td>
<td>23</td>
<td>USA (Georgia)</td>
</tr>
<tr>
<td></td>
<td>Southern bean mosaic</td>
<td>1-40</td>
<td>34,55,72</td>
<td>Africa, Americas</td>
</tr>
<tr>
<td></td>
<td>Sunn-hemp mosaic</td>
<td>4-20</td>
<td>52</td>
<td>Africa, India, US A</td>
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<tr>
<td></td>
<td>Urdbean leaf crinkle</td>
<td>6-15</td>
<td>9</td>
<td>India</td>
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* Detected in small plantings and eradicated  ** Data on rate of seed transmission not available
References
Table 2. Legume hosts in which natural seed-transmission has been reported.

<table>
<thead>
<tr>
<th>Virus</th>
<th>Host</th>
<th>% transmission</th>
<th>Reference</th>
<th>Geographical distribution</th>
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<tbody>
<tr>
<td>Alfalfa mosaic</td>
<td><em>Medicago polymorpha</em></td>
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<tr>
<td></td>
<td><em>M. sativa</em></td>
<td>1-30</td>
<td>66</td>
<td>Worldwide</td>
</tr>
<tr>
<td></td>
<td><em>M. truncatula</em></td>
<td>2</td>
<td>46</td>
<td>Australia</td>
</tr>
<tr>
<td>Bean common mosaic</td>
<td><em>Macroptilium lathyroides</em></td>
<td>5-33</td>
<td>68</td>
<td>Guyana, Hawaii, Philippines, Suriname</td>
</tr>
<tr>
<td></td>
<td><em>Phaseolus acutifolius</em></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>var. <em>latifolius</em></td>
<td>7-22</td>
<td>69</td>
<td>USA</td>
</tr>
<tr>
<td></td>
<td><em>P. vulgaris</em></td>
<td>0-83</td>
<td>60, 62</td>
<td>Worldwide</td>
</tr>
<tr>
<td></td>
<td><em>Vigna mungo</em></td>
<td>2-10</td>
<td>3</td>
<td>India</td>
</tr>
<tr>
<td></td>
<td><em>V. radiata</em></td>
<td>8-32</td>
<td>49</td>
<td>Iran</td>
</tr>
<tr>
<td>Bean pod mottle</td>
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<td>0.08</td>
<td>56</td>
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<tr>
<td>Bean yellow mosaic</td>
<td><em>Lupinus luteus</em></td>
<td>6-14</td>
<td>7-21</td>
<td>Worldwide</td>
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<tr>
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<td><em>Pisum sativum</em></td>
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<td>26</td>
<td>Worldwide</td>
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<tr>
<td></td>
<td><em>Viciafaba</em></td>
<td>0.1-2.4</td>
<td>48, 63</td>
<td>Iran, Sudan</td>
</tr>
<tr>
<td>Blackeye cowpea mosaic</td>
<td><em>Vigna unguiculata</em></td>
<td>30</td>
<td>83</td>
<td>Worldwide</td>
</tr>
<tr>
<td>Blackgram mottle</td>
<td><em>Vigna mungo</em></td>
<td>8</td>
<td>70</td>
<td>India, Thailand</td>
</tr>
<tr>
<td>Broad bean mottle</td>
<td><em>Viciafaba</em></td>
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<td>58</td>
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<td>1-10</td>
<td>32, 44, 45</td>
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<td>Syria</td>
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<td>Broad bean true mosaic</td>
<td><em>Viciafaba</em></td>
<td>1-17</td>
<td>11, 12, 20, 44</td>
<td>Australia*, Europe, northwest Africa</td>
</tr>
<tr>
<td>Cowpea aphid-borne mosaic</td>
<td><em>Vigna unguiculata</em></td>
<td>0-40</td>
<td>1, 50</td>
<td>Worldwide</td>
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<td>Virus</td>
<td>Host</td>
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<td>Reference</td>
<td>Geographical distribution</td>
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<td>-----------</td>
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<tr>
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<td>Vigna unguiculata</td>
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<td>17,77</td>
<td>Worldwide</td>
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<tr>
<td>Cowpea mosaic</td>
<td>Vigna unguiculata</td>
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<td>33</td>
<td>Cuba, Kenya, Nigeria, Suriname, USA</td>
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<td>Vigna unguiculata</td>
<td>0.2-10</td>
<td>5.73</td>
<td>Nigeria</td>
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<tr>
<td>Cowpea ringspot</td>
<td>Vigna unguiculata</td>
<td>10-30</td>
<td>67</td>
<td>Iran</td>
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<td>Cowpea severe mosaic</td>
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<td>40.71</td>
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<td>8</td>
<td>22</td>
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<td>82</td>
<td>China</td>
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<tr>
<td></td>
<td>Glycine max</td>
<td>30-100</td>
<td>74</td>
<td>Indonesia, Japan, USA, USSR</td>
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<tr>
<td></td>
<td>Lupinus albus</td>
<td>**</td>
<td>35</td>
<td>Europe</td>
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<tr>
<td></td>
<td>L. angustifolius</td>
<td>3-34</td>
<td>4.47</td>
<td>Australia</td>
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<td>L. luteus</td>
<td>21</td>
<td>84</td>
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<td>Phaseolus vulgaris</td>
<td>0-7</td>
<td>14.59</td>
<td>Worldwide</td>
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<td>Vigna radiata</td>
<td>10</td>
<td>42</td>
<td>Japan</td>
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<td></td>
<td>V. unguiculata</td>
<td>15-20</td>
<td>67</td>
<td>Worldwide</td>
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<td>Desmodium mosaic</td>
<td>Desmodium canum</td>
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<td>Guar symptomless</td>
<td>Cyamopsis tetragonoloba</td>
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<tr>
<td>Lucerne transient streak</td>
<td>Melilotus albus</td>
<td>2.5</td>
<td>10.65</td>
<td>Australia, Canada, New Zealand</td>
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<td>Lens culinaris</td>
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<td>37.38</td>
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<td>Pisum sativum</td>
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<tr>
<td></td>
<td>Viciafaba</td>
<td>**</td>
<td>64</td>
<td>Europe</td>
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Table 2. Legume hosts in which natural seed-transmission has been reported (cont'd),

<table>
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<tr>
<th>Virus</th>
<th>Host</th>
<th>% transmission</th>
<th>Reference</th>
<th>Geographical distribution</th>
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</thead>
<tbody>
<tr>
<td>Pea early-browning</td>
<td><em>Pisum sativum</em></td>
<td>1-37</td>
<td>15,41</td>
<td>Europe, Morocco</td>
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<td></td>
<td><em>Vicia faba</em></td>
<td>1-10</td>
<td>28, 29</td>
<td>Europe</td>
</tr>
<tr>
<td>Pea mild mosaic</td>
<td><em>Pisum sativum</em></td>
<td>15</td>
<td>19</td>
<td>New Zealand</td>
</tr>
<tr>
<td>Peanut clump</td>
<td><em>Arachis hypogaea</em></td>
<td>6-14</td>
<td>76</td>
<td>Burkina Faso, Côte d’Ivoire, India, Niger, Senegal</td>
</tr>
<tr>
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<td><em>Arachis hypogaea</em></td>
<td>0-20</td>
<td>2-13</td>
<td>Probably worldwide</td>
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<td></td>
<td><em>Lupinus albus</em></td>
<td>0.4</td>
<td>24</td>
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<tr>
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<td><em>Phaseolus vulgaris</em></td>
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<td>8</td>
<td>Australia (Queensland)</td>
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<td></td>
<td><em>Vigna unguiculata</em></td>
<td>&lt;1</td>
<td>23</td>
<td>USA (Georgia)</td>
</tr>
<tr>
<td>Peanut stripe</td>
<td><em>Arachis hypogaea</em></td>
<td>0.1-10</td>
<td>25</td>
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<td><em>Arachis hypogaea</em></td>
<td>0.1</td>
<td>78</td>
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</tr>
<tr>
<td>Southern bean mosaic</td>
<td><em>Phaseolus vulgaris</em></td>
<td>1-30</td>
<td>43,61</td>
<td>Africa, Americas, India</td>
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<tr>
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<td><em>Vigna unguiculata</em></td>
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<td>34,55,72</td>
<td>Africa, Americas</td>
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<td>Soybean mosaic</td>
<td><em>Glycine max</em></td>
<td>0.1-30</td>
<td>16,36</td>
<td>Worldwide</td>
</tr>
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<td>Soybean stunt</td>
<td><em>Glycine max</em></td>
<td>0-50</td>
<td>54</td>
<td>Japan</td>
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<tr>
<td>Subterranean clover mottle</td>
<td><em>Trifolium subterraneum</em></td>
<td>3</td>
<td>30</td>
<td>Australia</td>
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<tr>
<td>Sunn-hemp mosaic</td>
<td><em>Crotalaria juncea</em></td>
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<td>80</td>
<td>India</td>
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<td></td>
<td><em>Vigna catjang</em></td>
<td>17</td>
<td>18</td>
<td>India</td>
</tr>
<tr>
<td></td>
<td><em>V. unguiculata</em></td>
<td>4-20</td>
<td>52</td>
<td>Africa, Australia, India, USA</td>
</tr>
</tbody>
</table>
Table 2. Legume hosts in which natural seed-transmission has been reported (cont’d).

<table>
<thead>
<tr>
<th>Virus</th>
<th>Host</th>
<th>% transmission</th>
<th>Reference</th>
<th>Geographical distribution</th>
</tr>
</thead>
<tbody>
<tr>
<td>Tobacco ringspot</td>
<td><em>Glycine max</em></td>
<td>0-100</td>
<td>6</td>
<td>Egypt, India, North America, Sri Lanka, Turkey</td>
</tr>
<tr>
<td>Tobacco streak</td>
<td><em>Glycine max</em></td>
<td>0-90</td>
<td>31-79</td>
<td>Argentina, Brazil, USA</td>
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<td></td>
<td><em>Melilotus albus</em></td>
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<td>75</td>
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<td>China</td>
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<tr>
<td>Urdbean leaf crinkle</td>
<td><em>Vigna mungo</em></td>
<td>18</td>
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<td>India</td>
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<tr>
<td></td>
<td><em>V. unguiculata</em></td>
<td>6-15</td>
<td>9</td>
<td>India</td>
</tr>
</tbody>
</table>

* Detected in small plantings and eradicated

** Data on rate of seed transmission not available
   Phytopathology 48:86-91.


Most of the pathogens listed in Table 3 are carried internally and externally on the seed. They may also be carried with the seed in contaminated dust, crop debris or soil. The latter method is probably one of the means by which *Pseudomonas solanacearum* is disseminated, but the frequency of transmission is likely to be extremely low and its importance is uncertain. For the majority of the pathogens, seed-borne inoculum is of major importance to their survival and dissemination.

**Quarantine measures and seed health testing**
Levels of bacterial infection in seed stocks are often low and range from < 0.01% to 1% (1% is considered a high level for a bacterial disease). The transmission from seed to seedling is also relatively inefficient (about 1 out of 10). It follows that very large amounts of seed would be necessary to detect infection by growing-on tests. Moreover, in the glasshouse, conditions may be unfavourable for disease expression and infected plants may remain symptomless. Because of this, laboratory seed tests are preferred. These methods involve extraction of bacteria from seed by soaking or macerating. The bacteria are then either isolated on agar medium, with or without selective agents, or detected by indirect serological methods; immunofluorescence (IF) or enzyme-linked immuno-sorbent assay (ELISA). The agar isolation procedure has some advantages: it is potentially highly sensitive ($10^2$ bacterial cells per ml seed extract) and it may be linked to a variety of identification techniques such as cultural and biochemical tests, bacteriophage, serology (agglutination, gel diffusion, IF, ELISA) and host inoculation (leaves, pods, stems).

Many of the detection methods have recently been assembled (Saettler *et al.*, 1989) and general identification techniques suitable for all the pathogens mentioned are given by Lelliot & Stead (1987) and Schaad (1980). The currently available seed tests are particularly appropriate to the pathovars of *Pseudomonas syringae* and *Xanthomonas campestris*. Serological methods may not distinguish between some of the pathovars, especially the pathovars of *X. campestris*. With considerable overlap in their host range there is some doubt as to their distinctness.

Antibiotic seed treatments have shown some promise in reducing both internal and external seed infection (Taylor & Dudley, 1977; Taylor & Dye, 1976). However, disease control is not completely effective and antibiotics are not generally permitted on crops destined for food. Treatment of seeds with short soaks (1-5 mins) or dips in sodium hypochlorite (1-2% available chlorine) will reduce both surface infection and contamination by infected dust or debris.
For the safe movement of legume germplasm a combination of methods should be considered.

- Multiply small seed samples under containment and harvest seed only from healthy looking plants.
- Surface sterilize seed with sodium hypochlorite or other chlorine containing compound.
- Apply laboratory seed tests if available.

References


Table 3. Seed-borne bacterial pathogens of grain legumes.

<table>
<thead>
<tr>
<th>Bacterium</th>
<th>Principal leguminous host</th>
</tr>
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<tbody>
<tr>
<td><em>Clavibacter</em> michiganense subsp. insidiosum</td>
<td>alfalfa / lucerne</td>
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<tr>
<td><em>Curtobacterium</em> flaccumfasciens pv. flaccumfasciens</td>
<td>bean</td>
</tr>
<tr>
<td><em>Pseudomonas</em> solanacearum</td>
<td>groundnut</td>
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<td><em>Pseudomonas</em> syringae</td>
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<td>pea</td>
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<td>pv. syringae</td>
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<tr>
<td>pv. tabaci</td>
<td>soybean</td>
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<tr>
<td><em>Xanthomonas campestris</em></td>
<td></td>
</tr>
<tr>
<td>pv. alfalfa</td>
<td>alfalfa / lucerne</td>
</tr>
<tr>
<td>pv. cajani</td>
<td>pigeonpea</td>
</tr>
<tr>
<td>pv. cassiae</td>
<td>chickpea</td>
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<tr>
<td>pv. cyamopsidis</td>
<td>clusterbean</td>
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<tr>
<td>pv. glycinea</td>
<td>soybean</td>
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<td>pv. phaseoli</td>
<td>common bean</td>
</tr>
<tr>
<td>pv. pisi</td>
<td>pea</td>
</tr>
<tr>
<td>pv. vignaeradiatae</td>
<td>mungbean</td>
</tr>
<tr>
<td>pv. vignicola</td>
<td>cowpea</td>
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</tbody>
</table>

* formerly *Corynebacterium*
1. Bacterial blight of pea

Cause
*Pseudomonas syringae* pv. *pisi* (Sackett) Young, Dye & Wilkie.

Symptoms
The disease affects all above-ground parts (stems, leaves, pods and tendrils). Lesions, at first water-soaked, become brown and necrotic. Infected seeds may be shrivelled or show olive green patches, they may also be symptomless.

Geographical distribution
Widespread (Anonymous, 1971).

Host range
*Lathyrus* spp., *Pisum sativum*. Isolates of the pathovar are categorised into at least 6 races on the basis of the reactions of a range of differential pea cultivars (Taylor *et al.*, 1989).

Biology and transmission
Seed transmitted externally or internally in *Pisum sativum* (Skoric, 1927; Sutton & Katznelson, 1953; Close, 1966; Watson & Dye, 1971).

References
2. **Bacterial blight of soybean**

**Cause**  
*Pseudomonas syringae* pv. *glycinea* (Coerper) Young, Dye & Wilkie.

**Symptoms**  
Small leaf spots, initially water soaked, becoming brown and necrotic, surrounded by yellow halos. Lesions may enlarge and coalesce, causing extensive necrosis. Lesions may also occur on stems and pods.

**Geographical distribution**  
Worldwide (Bradbury, 1986).

**Host range**  
*Glycine max*, *Glycine* spp. and possibly a number of *Phaseolus* spp. Isolates of the pathovar are categorised into 9 races on the basis of the reactions of a range of differential soybean cultivars (Cross *et al.*, 1966; Thomas & Leary, 1980; Fett & Sequeira, 1981).

**Biology and transmission**  

**References**  

3. **Bacterial brown spot**

**Cause**  
*Pseudomonas syringae* pv. *syringae* van Hall.
Symptoms
Brown spots on leaves and pods, shrivelled seeds.

Geographical distribution

Host range
Many important legume and non-leguminous crops.

Biology and transmission

References

4. Bacterial wilt

Cause
*Pseudomonas solanaceous* (Smith) Smith.
Divided in 4 biovars (Hayward, 1964), 13 ‘pathotypes’ (Okabe & Goto, 1961) and 3 races (Buddenhagen *et al.*, 1962). The latter based on their host range on important solanaceous hosts.
Symptoms
Systemic infection of the vascular system causes wilting as the main symptom, with or without browning of vascular tissues, bacterial exudate from cut vessels, stunting and chlorosis of plants.

Geographical distribution

Host range
Very wide host range mainly non-legumes but including important legumes such as Arachis hypogaea, Glycine max, Lablab purpureus, Medicago sativa, Phaseolus vulgaris, Pisum sativum, Psophocarpus tetragonolobus, Vicia faba, Vigna radiata and V. unguiculata.

Biology and transmission
Occasionally seed-borne in soybean (Muras, 1964) and in groundnut (Palm, 1922).

References

5. Bacterial wilt of bean

Cause
*Curtobacterium flaccumfaciens* pv. *flaccumfaciens* (Hedges) Collins & Jones.

Symptoms
Seedlings are stunted, wilted and usually die. Older plants wilt, show a dull green of affected parts, and sometimes breaking of the stems. Infected pods show discoloured sutures and may show yellowish areas.
**Geographical distribution**
Australia, Belgium, Bulgaria, Canada, Colombia, Greece, Hungary, Mexico, Rumania, Tunisia, Turkey, USA, USSR, Yugoslavia. (Anonymous, 1987).

**Host range**
*Lablab purpureus, Phaseolus coccineus, P. lunatus, P. vulgaris, Vigna angularis, V. unguiculata, Zornia spp.* (cover crops), and possibly in *Glycine max* (Bradbury, 1986). All members of the Leguminosae.

**Biology and transmission**
Seed transmitted externally or internally in *Phaseolus vulgaris* and possibly in *Glycine max* (Leonard, 1924; Burkholder, 1926; Dunleavy, 1962). The pathogen can survive from 5-24 years in seed (Schuster & Coyne, 1974).

**References**

**6. Bacterial wilt of lucerne**

**Cause**
*Clavibacterium michiganensis* subsp. insidiosum McCulloch, Davis, Gillaspie, Vidaver & Harris.

**Symptoms**
Stunted plants, yellowed with darkened vascular tissues in the roots. Plants may be killed the second year after infection.
**Geographical distribution**
Australia, Brazil, Britain, Canada, Czechoslovakia, Italy, Mexico, New Zealand, Poland, Saudia Arabia, South Africa, USA, USSR (Anonymous, 1987).

**Host range**
The main natural host is *Medicago sativa*; also reported to occur naturally on *Lotus corniculatus*, *Medicago falcata*, *Melilotus alba*, *Onobrychis viciaefolia* and *Trifolium* sp.

**Biology and transmission**
Seed transmission in *Medicago sativa* both by seed and by debris mixed with seed (Cormack & Moffatt, 1955; Cormack, 1961; Golenia, 1965).

**References**

**7. Common bacterial blight of bean**

**Cause**
*Xanthomonas campestris* pv. *phaseoli* (Smith) Dye.

**Symptoms**
On leaves, initially small water soaked lesions develop narrow, yellow halos. Lesions may enlarge and coalesce, causing extensive necrosis. Lesions may also occur on stems and pods. Infected seeds are sometimes wrinkled and the hilum may be discoloured. Symptoms similar to halo-blight of bean.

**Geographical distribution**
Very widespread (Anonymous, 1971).
**Host range**

*Macroptilium lathyroides, Phaseolus lunatus, P. vulgaris* and the weed *Strophostyles helvola*. *Lablab purpureus* is reported as a natural host but most references involve inoculation. Special races or strains are reported to occur naturally on *Phaseolus aconitifolius* in India, on *Vigna umbellata (Phaseolus calcaratus)* and *V. radiata (P. aureus)* in China and on *V. mungo* in India.

**Biology and transmission**


**References**


**8. Halo blight of bean**

**Cause**

*Pseudomonas syringae pv. phaseolicola* (Burkholder) Young, Dye & Wilkie.

**Symptoms**

Small leaf spots, initially water soaked, becoming brown and necrotic, surrounded by broad yellow chlorotic halos. Chlorosis is due to a toxin produced by the bacterium. Toxin may be translocated producing virus-like interveinal chlorosis and distortion of leaves even in the absence of lesions. Lesions on stems and pods are also water soaked, sometimes with bacterial exudate. Pod lesions have the appearance of ‘grease’ spots. Seeds from infected pods may be shrivelled and wrinkled. White-seeded varieties may show buttery yellow patches on the seed coat but infected seed may also be symptomless.
Geographical distribution
Worldwide (Anonymous, 1973). In temperate climatic conditions and in the tropics at medium to high altitudes (1000-2500 m). Race 3 of the pathogen has been found only in East and Central Africa.

Host range
Isolates of the pathovar are categorised into three races on the basis of the reactions of a range of differential bean cultivars (Taylor et al., 1987).

Biology and transmission
Seed transmitted externally or internally in Phaseolus vulgaris (Burkholder, 1926; Katznelson et al., 1954; Grogan & Kimble, 1967; Taylor, 1970) and probably all other hosts.

References
Fungal diseases

1. Angular leaf spot of kidney bean (*Phaseolus vulgaris*)

**Cause**

*Phaeoisariopsis griseola* (Sacc.) Ferraris  
Synonym: *Isariopsis griseola* Sacc.

**Symptoms**

Reddish brown lesions on leaves with typical angular margins. Sporulation under continuous moisture for 24-48 h. Circular or irregular spots on stem, petioles, branches and pods.

**Geographical distribution**

Widespread (Anonymous, 1986a; 1986b).

**Alternative hosts**

*Desmodium cephalotus, D. gangeticum, D. pulchellum, Dolichos lablab, Phaseolus lunatus, P. multiflorus, Pisum sativum, Vigna unguiculata.*

**Biology and transmission**

Seed transmitted (Orogoco-Sarria & Cordona Alvarez, 1959) and through plant debris. Rain splash and wind help in disease spread. Seed infected at the hilum region (Sohi & Sharma, 1974).

**Quarantine measures and seed health testing**

- The fungus can be detected by incubating seeds on either agar or wet blotters at 24°C (Orogoco-Sarria & Cordona Alvarez, 1959).
- Seed treatment with 0.2% benomyl powder controlled the disease (Bose & Sindhan, 1972).
- Storing seeds for over one year kills the fungus completely.

**References**


2. Ascochyta blight of chickpea

Cause
Ascochyta rabiei (Pass.) Labrousse;
   perfect state: Mycosphaerella rabiei (Pass.) Kovach.
Two races are reported from India (Vir & Grewal, 1975).

Symptoms
All aerial parts are affected. Brown to dark brown elongated lesions on stem, and dark brown on leaves, with sunken tissue and dark margins. Pycnidia can sometimes be observed in the affected tissues.

Geographical distribution
Algeria, Australia, Bangladesh, Bulgaria, Canada, Cyprus, Ethiopia, France, Greece, India, Iran, Iraq, Israel, Italy, Lebanon, Mexico, Morocco, Pakistan, Romania, Spain, Syria, Tunisia, Turkey, USA and USSR.

Alternative hosts
Not known.

Biology and transmission
Seed transmitted, but plant debris also play an important role in transmission. Mycelium is present in seed coat and cotyledons (Maden et al., 1975).

Quarantine measures and seed health testing
- The pathogen can be detected by two methods:
  * Plate seeds directly on water-soaked blotters, incubate at 22°C for 7 days under 12 hours photoperiod of NUV or artificial daylight (Mathur, 1981). Look for pycnidia and characteristic pycnospores.
  * Plate surface sterilized seeds on PDA containing 1 g dicrysticin/litre and incubate at 20°C for 8 days under 12 hours photoperiod of NUV or artificial daylight (Haware et al., 1986). Creamy fungus colonies with black centre.
- Seed treatment with tridemorph alone or in mixture with benomyl gives complete control (Reddy, 1980).

References
3. **Bean anthracnose**

**Cause**

*Colletotrichum lindemuthianum* (Sacc. & Magn.) Bri. & Cav.

Three races have been reported (Cruickshank, 1966).

**Symptoms**

Symptoms can appear on any plant part. Rust-coloured specks on cotyledons, brick-red to purple or black lesions on petiole, leaves and leaf veins. Brown sunken cankers delimited by black rings on pods. Lesions on seeds are brown with white centre, or reddish.

**Geographical distribution**

Africa, Asia, Australia, Brazil, Colombia, Costa Rica, Europe, Guatemala, Mexico, North America, Venezuela.

**Alternative hosts**

*Phaseolus* spp., *Vigna* spp., *Vicia* spp. and many other plant species.

**Biology and transmission**

Infected seeds (cotyledons and seed coat) and plant debris are the primary sources of inoculum. Intermittent moderate rainfall and temperature between 13 and 26°C are conducive for spread.

**Quarantine measures and seed health testing**

- The pathogen can be detected by two methods:
  - Pretreat the seeds for 10 min in sodium hypochlorite solution (1% available chlorine) and plate on wet blotters, incubate for 7 days at 20°C in darkness (Anselme & Champion, 1981).
  - Growing-on test in sand at room temperature for 14 days (Kummer & Schmidt, 1961).
- Seed treatment with benomyl (Sindhan & Bose, 1981) and Orthocide (Petrov, 1972) give best control.
References

4. Brown spot of soybean

Cause
Septoria glycines Hemmi.

Symptoms
Irregular dark-brown spots on leaves, stem, branches, petioles and pods. Leaves turn yellow and drop.

Geographical distribution
Brazil, Canada, China, Germany, Italy, Japan, Korea, Taiwan, USSR and Yugoslavia.

Alternative hosts
Not known.

Biology and transmission
Infected seeds (mycelium in seed coat), leaves and plant debris are the sources of primary inoculum. The lesions produced on young plants act as secondary sources when the weather is warm and wet and the inoculum is distributed by wind and rain splashes. Dry weather is inhibitory (Sinclair & Backman, 1989).

Quarantine measures and seed health testing
- Although no standard testing method has been established, the fungus can be detected by plating seeds on wet blotters and incubating under light (12 hours daily) for 7 days.

Reference
5. Charcoal rot of groundnut

Cause
*Macrophomina phaseolina* (Tassi.) G. Goid.

Synonyms: *Macrophomina phaseoli* (Maub.) Ashby.

*Sclerotium bataticola* Taub.

Pycnidial stage of *Rhizoctonia bataticola* (Taub.) Butl.

Various strains of *M. phaseolina* are reported to occur in nature.

Symptoms
Seed and seedling rots, root and stem rots, rotting of developing pods and seeds. The tap root turns black and later becomes rotten, shredded, and studded with sclerotia. Pods are also attacked, the pathogen rapidly infects the fruits, developing symptoms of blacknuts and leading also to concealed damage. Infected seeds are discoloured, small, shrivelled, and have a dirty black appearance. Severely attacked seeds are covered with a profuse growth of the mycelium of the fungus on the inner as well as on the outer surfaces of the two cotyledons, and black sclerotia can be observed in the endosperm tissue. Some infected seeds do not show external symptoms.

Geographical distribution
Argentina, Gambia, India, Israel, Nigeria, Senegal, USA and Venezuela.

Alternative hosts
Found throughout the world, causing diseases in a large number of crop species.

Biology and transmission
Charcoal rot is both seed-borne and soil-borne. Mycelium in seeds and sclerotia in plant debris in the soil are primary sources of inoculum. The fungus persists in the soil for long periods either as actively growing mycelium or as dormant sclerotia. The pathogen is commonly present in groundnut seeds (mycelium in cotyledons or endosperm) and pods, and can readily be disseminated by their movement. Mycelial fragments as well as sclerotia can be present on the testae of seeds.

Quarantine measures and seed health testing
- The pathogen can be detected by two methods:
  * Pretreat the seeds for 10 min in sodium hypochlorite solution (1% available chlorine) or for 2 min in a 0.1% aqueous solution of mercuric chloride, plate on wet blotters and incubate at 25°C in darkness.
  * Plate seeds on to potato dextrose agar in petri plates and then incubate at 25°C in the dark for 5-7 days. Surface-sterilized cut pieces of seeds can also be tested for seed-borne infection. To obtain quick results, plating on agar is preferred.
• Seed treatment with fungicides such as Quintozene (PCNB) and Captan completely eradicates seed-borne infection without any adverse effect on seed germination.

References

6. Downy mildew of soybean

**Cause**
*Peronospora manshurica* (Naum.) Syd.
There are 32 known races (Sinclair & Backman, 1989).

**Symptoms**
Pale green to pale yellow spots on the upper leaf surface, turning brown to dark brown with yellow margins. On lower surface grey to purple-coloured conidiophores in moist weather. Symptoms may not appear on pods, which may contain white mycelium on seeds. Infected seeds are small and encrusted with mycelium and oospores.

**Geographical distribution**

**Alternative hosts**
Not known.

**Biology and transmission**
Systemically transmitted to seedlings (Novakova & Pfeifferova, 1964). Infected seeds and plant debris are the primary sources of inoculum. Mycelium and oospores can be found on seed, and mycelium in the seed coat.

**Quarantine measures and seed health testing**
• Examination of seed washings (Hansen & Mathur, 1987).
• Seed treatment is only partly effective.
7. **Early leafspot of groundnut**

**Cause**
*Cercospora arachidicola* Hori.

Synonyms: *Mycosphaerella arachidicola* W.A. Jenkins
*Mycosphaerella arachidis* Deighton

There is some evidence of variation between isolates of the pathogen, but the pathotypes have not been clearly characterized.

**Symptoms**
Subcircular lesions, dark brown on the upper leaflet surface where most sporulation occurs, and light brown on the lower leaflet surface. When attack is severe, the affected leaflets first become chlorotic and then necrotic, lesions often coalesce, and leaflets are shed. In addition to leaf spots, lesions are also produced on petioles, stems and pegs.

**Geographical distribution**
Commonly present wherever groundnut is grown (Anonymous, 1985).

**Alternative hosts**
Some members of the genus *Arachis*. There is no record of any hosts outside the genus *Arachis*.

**Biology and transmission**
The principal source of initial inoculum is probably conidia produced on groundnut crop residues in the soil. Inoculum is blown or splashed on to leaves giving rise to primary infection. Conidia are disseminated by wind, rain splash and insects leading to secondary infection. The pathogen may also survive on volunteer groundnut plants and on groundkeepers. Long distance spread may be by movement of infected crop debris, pods or seeds externally contaminated with conidia. There is no evidence of the disease being internally seed-borne. The role of seed-borne inoculum on disease spread is not known.

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**References**


Quarantine measures and seed health testing
- Seed treatment with carbendazin has been recommended to eradicate externally seed-borne inoculum.
  Information on seed health testing is not available.

References

8. Groundnut rust

Cause
*Puccinia arachidis* Speg.

Synonyms: *Uredo arachidis* Lagerheim
*Uromyces arachidis* P. Hennings
*Bullaria (?) arachidis* (Speg) Arthur & Mains.

Symptoms
Orange-coloured pustules (uredinia) observed on lower surface of leaf, but with advance of disease, they can be seen on upper surface and other aerial parts except flowers and pegs. Rusted leaves tend to remain attached to the plant.

Geographical distribution
Almost all groundnut growing areas of the world (Subrahmanyam *et al.*, 1984; Anonymous, 1985).

Alternative hosts
Some species of *Arachis*.

Biology and transmission
Long-distance dissemination may be by air-borne urediniospores, infected crop debris, or pods or seeds externally contaminated with urediniospores. Seed-borne inoculum may play a role in disease transmission (Peregrine, 1971) but according to Subrahmanyam & McDonald (1982) and Subrahmanyam *et al.* (1984) there is no
evidence that peanut rust is seed transmitted. However, rust spores present on the seed surface or in packing material may become a source of primary infection if released during handling.

Quarantine measures and seed health testing
- Avoid movement of pods.
- Packing material should be carefully inspected for the presence of urediniospores upon arrival and a washing test performed (examination of seed washings).
- Seeds should be treated with appropriate fungicide (Varma & McDonald, 1984).

References

9. Groundnut scab

Cause
*Sphaceloma arachidis* Bit. & Jenk.

Symptoms
Small chlorotic spots, spread uniformly or in clusters near the veins, on both sides of the leaves. Spots on upper surface later become tan with raised margins, while those on lower surface are darker and not raised. The maximum size of spots is less than 2 mm. On stem and petioles, the growth is corky, giving the plant a burned appearance. The fungus produces fructifications under high humidity.

Geographical distribution
Argentina, Brazil, Colombia and Japan.

Alternative hosts
Apparently restricted to the genus *Arachis*. 
Biology and transmission
The fungus persists in crop debris that acts as source of inoculum. There is some evidence of possible seed transmission (Giorda, 1984) but these observations were not substantiated by further work.

Quarantine measures and seed health testing
- Incubate seeds on wet blotters or agar for 7 days at 22°C ±2°C under alternating cycles of 12 hours of light from NUV and darkness.
- Foliar application of benomyl is effective in controlling the disease but its efficacy as a seed treatment is not known.

References

10. Late leafspot of groundnut

Cause
Phaeoisariopsis personata (Berk. & Curt.) v. Arx
Synonyms:
- Cercosporidium personatum (Berk. & Curt.) Deighton
- Cladosporium personata Berk. & Curt.
- Cercospora personata (Berk. & Curt.) Ellis & Everhart
- Passalora personata (Berk. & Curt.) Khan & Kamal
- Septogloeum arachidis Racibolski
- Mycosphaerella berkeleyii W.A. Jenkins

Symptoms
Lesions are dark, usually small and nearly circular. On the lower surfaces, where most sporulation occurs, the lesions are black with a slightly rough appearance. When attack is severe, the affected leaflets first become chlorotic, then necrotic, lesions often coalesce, and leaflets are shed. In addition to leaf spots, the pathogen also produces lesions on petioles, stems and pegs.

Geographical distribution
Almost all groundnut-growing areas of the world (Anonymous, 1987).

Alternative hosts
Some members of the genus Arachis. There is no record of any host outside the genus Arachis.

Biology and transmission
Same as early leafspot.
Quarantine measures and seed health testing
- Seed treatment with carbendazin has been recommended to eradicate externally seed-borne inoculum.
- Information on seed health testing is not available.

References

11. Pepper spot and leaf scorch of groundnut

Cause
Leptosphaerulina crassiasca (Sechet) Jackson & Bell
Synonyms: Pleospora crassiasca Sechet
Leptosphaerulina arachidicola Yen, Chen & Huang
Pleospora arachidicola Huang
Leptosphaerulina trifolii (Rest.) Petr.
Pseudoplea trifolii (Rost.) Petr.

Symptoms
Dark brown to black discrete lesions on both sides of the leaflets. When lesions are abundant, they tend to coalesce giving the leaflet surface a netted appearance. In such cases leaflets soon die and production of numerous ascocarps occurs in necrotic areas of abscised leaflets. Leaf scorch symptoms frequently develop on the tips of leaflets, forming a wedge-shaped lesion with a bright yellow zone along the periphery of the advancing margin of the lesion. Ascocarps of the fungus are abundant in the dead tissue.

Geographical distribution
Argentina, Burkina Faso, China, India, Madagascar, Malawi, Mauritius, Niger, Nigeria, Senegal, Taiwan, USA and Vietnam. The disease is probably present in several other groundnut-growing countries.

Alternative hosts
Apparently restricted to the genus Arachis.
Biology and transmission
An asexual stage of the fungus is unknown. Ascocarps are produced abundantly in infected leaf debris. The longevity of the pathogen and the mode of spread of the disease are not known.

Quarantine measures and seed health testing
Not known.

References

12. Soybean root and stem rot

Cause
Phytophthora megasperma Drechsler var. sojae Hildebrand
Twenty races are known (Keeling, 1982).

Symptoms
The fungus can attack soybean at any stage of growth and can cause seed rot and pre-emergence damping-off. In young plants, stem appears water-soaked, leaves turn yellow and ultimately the plant dies. In mature plants leaves become chlorotic and droop due to fungal infection in vascular bundles.

Geographical distribution
Australia, Canada, USA.

Alternative hosts
Lupinus spp., tomato, alfalfa, garden pea, snap bean (Phaseolus vulgaris), Trifolium subterraneum and T. repens.

Biology and transmission
Primary inoculum, chiefly as oospores, comes from crop residues in the soil where the fungus survives long periods in the absence of soybean crops. The pathogen is also transmitted by seed and by soil mixed with seed.

Quarantine measures and seed health testing
• No specific test is described in literature. A selective medium developed by Keeling (1980) may be used to isolate the fungus from seed. The ingredients of the medium are 40 ml of V-8 juice, 0.6 g of calcium carbonate, 0.2 g of yeast extract, 1 g of sucrose, 10 mg of cholesterol, 20 mg of 50% benomyl, 27 mg of pentachloronitrobenzene, 100 mg of neomycin sulphate, 30 mg of chloramphenicol and 20 g of agar in 1 litre of water.
• Infusion of pyroxychlor, dissolved in acetone, into seed before planting is recom-
mended by Papavizas & Lewis (1976).

References
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Press, Boulder.
Keeling, B.L. 1982. Four new physiological races of Phytophthora megasperma f. sp.
Papavizas, G.C. & Lewis, J.A. 1976. Acetone infusion of pyroxychlor into soybean seed
for the control of Phytophthora megasperma var. sojae. Plant Dis. Repr 60: 484-
488.

13. Wilt of chickpea

Cause
Fusarium oxysporum Schlecht. emend. Snyd. & Hans. f. sp. ciceri (Padwick) Snyd. &
Hans.
Different pathogenic races are known.

Symptoms
The pathogen causes vascular, wilt in chickpea. Wilting can occur in seedling or adult
stages. The initial symptom is drooping of petioles and rachis along with leaflets.
Within 2 to 3 days, drooping is seen on the entire plant. Roots of wilted plants show
no external rotting, but when split vertically, clearly show internal discoloration of the
xylem.

Geographical distribution
Algeria, Bangladesh, Chile, Ethiopia, India, Iran, Italy, Lebanon, Malawi, Mexico,
Morocco, Myanmar (Burma), Pakistan, Peru, Spain, Sudan, Syria, Tunisia and the
USA.

Alternative hosts
Pigeonpea, pea and lentil are symptomless carriers of the pathogen.

Biology and transmission
The disease is transmitted to new areas through infected seed (chlamydospore-like
structures in the hilum region of the seed). Soil-borne inoculum is also a source of
primary infection. Once it is introduced to soil, it is difficult to eradicate the pathogen.
Therefore, it is important to stop spread of the pathogen through seed.
Quarantine measures and seed health testing
- Seeds are surface-sterilized by dipping for 2 min in 2.5% sodium hypochlorite and then plated on modified Czapek-Dox Agar, which contains, in addition to normal ingredients, 500 mg PCNB, 25 mg malachite green, 750 mg dicrysticin-S and 2 g yeast extract per litre of medium. The plates are inoculated at 20°C for 8 days in a cycle of 12 h NUV and 12 h of darkness. The white mycelium can then be seen emerging from infected seeds (Haware et al., 1978).
- Haware et al. (1978) demonstrated that a mixture of 30% benomyl and 30% thiram can completely eradicate seed-borne inoculum.

Reference

14. Wilt of pigeonpea

Cause
*Fusarium udum* Butler.

Symptoms
The pathogen causes vascular wilt in pigeonpea. The disease is characterized by gradual chlorosis followed by drying of the plant. Black streaks occur in the vascular region as well as under the bark in the lower part of the stem and tap root. Partial wilting of plants is common.

Geographical distribution
Widespread in Africa and India. It is reported from Bangladesh, Ghana, India, Indonesia, Kenya, Malawi, Mauritius, Nepal, Tanzania, Thailand, Trinidad and Uganda.

Alternative hosts
Not known.

Transmission
The pathogen is both seed-borne (mycelium present in the seed coat and cotyledons) and soil-borne. Once established in the soil, it is difficult to eradicate.

Quarantine measures and seed health testing
- Plating of pigeonpea seeds on Nash and Snyder’s medium. After incubation at 25°C for 10 days, mycelium growing out of infected seeds can be observed.
- Seed dressing with a mixture of benomyl 50 WP and thiram 75 WP (1:1) should eradicate the internal seed-borne *F. udum*. 
<table>
<thead>
<tr>
<th>Latin name</th>
<th>English</th>
<th>French</th>
<th>Spanish</th>
<th>German</th>
<th>Other</th>
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<tbody>
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<td>Arachis hypogaea</td>
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<td>arachide</td>
<td>mani</td>
<td>gemeine Erdnuss</td>
<td>kacang tanah</td>
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<td>Cajanus cajan</td>
<td>pigeonpea</td>
<td>pois d’Angole</td>
<td>guisante de paloma</td>
<td>Straucherbse</td>
<td>fèveJacques</td>
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<td>haba de burro</td>
<td>käcang parang</td>
<td>chana, Bengal gram</td>
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<td>guar, aconite bean</td>
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<td>almorta</td>
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<td>gesse commune</td>
<td>lenteja</td>
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<td>lupino</td>
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<td>grain de cheval</td>
<td>ojo de venado</td>
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<td>frijol trigo</td>
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<td>Texan bean</td>
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<td>haba Lima</td>
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<td>haricot commun</td>
<td>frijol</td>
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<tr>
<td>Trigonella foemina-graecum</td>
<td>fenugreek</td>
<td>fève</td>
<td>haba comun</td>
<td>Ackerbohne</td>
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<td>Vicia faba</td>
<td>faba bean</td>
<td>vesce commune</td>
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<td>ambérique</td>
<td>judia de urd</td>
<td>Urdbohne</td>
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<td>meth</td>
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<td>cowpea</td>
<td>pois vache</td>
<td>chicaro de vaca</td>
<td>Kuhbohne</td>
<td>take-azuki</td>
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</tbody>
</table>

* Table kindly provided by Dr L.J.G. van der Maesen, Department of Plant Taxonomy, Agricultural University, Wageningen, the Netherlands.
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