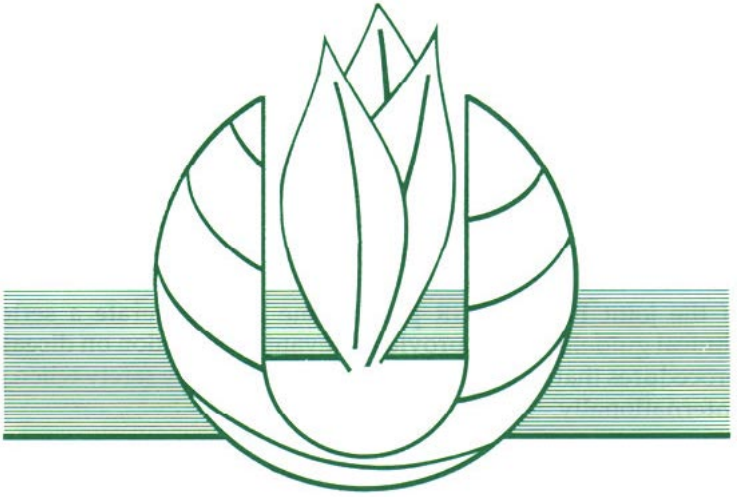
FOOD AND AGRICULTURE ORGANIZATION
OF THE UNITED NATIONS

INTERNATIONAL BOARD FOR
PLANT GENETIC RESOURCES

FAO/IBPGR TECHNICAL GUIDELINES
FOR THE

SAFE MOVEMENT OF LEGUME GERMPLASM



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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 re-quirements.
- 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

Beczner, L. & Manninger, S. 1975. Epidemiology of alfalfa mosaic virus, investigations on aphid transmission and seed transmission. Különlenyomat A Növényvédelmi Kutató Intézet Evkönyve **13**:167-176.

Hemmati, K. & McLean, D.L. 1977. Gamete-seed transmission of alfalfa mosaic virus and its effect on seed germination and yield in alfalfa. *Phytopathology* **67**:576-579

Jaspars, E.M.J. & BOS, L. 1980. Alfalfa mosaic virus. CMI/AAB Descriptions of Plant Viruses No. 229. Commonwealth Agricultural Bureaux, Slough.

Mishra, M.D., Raychaudhuri, S.P., Ghosh, A. & Wilcoxson, R.D. 1980. Berseem mosaic, a seed-transmitted disease. *Plant Dis.* **64**:490-492.

Pesic, Z. & Hiruki, C. 1986. Differences in the incidence of alfalfa mosaic virus in seed coat and embryo of alfalfa seed. *Can. J. Plant Pathol.* **8**:38-42.

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

Agarwal, V.K., Nene, Y.L., Beniwal, S.P.S. & Verma, H.S. 1979. Transmission of bean common mosaic virus through urdbean *(Phaseolus mungo)* seeds. *Seed Sci. & Technol.* **7**:103-108.

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Provvidenti, R. & Braverman, S.W. 1976. Seed transmission of bean common mosaic virus in phasemy bean. *Phytopathology* **66**:1274-1275.

Provvidenti, R. & Cobb, E.D. 1975. Seed transmission of bean common mosaic virus in tepary bean. *Plant Dis. Reptr* **59**:966- 969.

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
Lin, M.T. & Hill, J.H. 1983. Bean pod mottle virus: occurrence in Nebraska and seed transmission in soybeans. *Plant Dis.* **67**:230- 233.

Moore, B.J. & Walters, H.J. 1969. *Desmodium paniculatum,* a perennial host of bean pod mottle virus in nature. *Plant Dis. Reptr* **53**:154-155.

Semancik, J.S. 1972. Bean pod mottle virus. CMI/AAB Descriptions of Plant Viruses No. 108. Commonwealth Agricultural Bureaux, Slough.

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.l-2.4%: Kaiser, 1973; 0.l-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

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Quantz, L. 1954. Untersuchungen uber die Viruskrankheiten der Ackerbohne. *Mitt. Biol. Bundesanst. Land- u. Forstwirtsch.* (Berlin-Dahlem) **80**:171-175.

Zschau, K. 1962. Versuche und Beobachtungen zur Samenubertragung der Mosaikkrankheit der Lupinen, insbesondere der Gelblupine. *Nachr. Bl. Dtsch. Pfl.Sch.D.* **16**:1-7.

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

Anderson, C.W. 1955. *Vigna* and *Crotalaria viruses* in Florida II. Notations concerning cowpea mosaic *virus(Marmor vignae). Plant Dis. Reptr* **39**:349-353.

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Taiwo, M.A., Gonsalves, D., Provvidenti, R. & Thurston, H.D. 1982. Partial characterization and grouping of isolates of blackeye cowpea mosaic and cowpea aphid-borne mosaic viruses. *Phytopathology* **72**:509-596.

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Zettler, F.W. & Evans, I.R. 1972. Blackeye cowpea mosaic virus in Florida: host range and incidence in certified cowpea seed. *Proc. Fla St. Hort. Soc.* **85**:99-101.

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

Phatak, H.C. 1974. Seed-borne plant viruses - identification and diagnosis in seed health testing. *Seed Sci. & Technol.* **2**:3-155.

Scott, H.A. & Hoy, J.W. 1981. Blackgram mottle virus. CMI/AAB Descriptions of Plant Viruses No. 237. Commonwealth Agricultural Bureaux, Slough.

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

Gibbs, A.J. 1972. Broad bean mottle virus. CMI/AAB Descriptions of Plant Viruses No. 101. Commonwealth Agricultural Bureaux, Slough.

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Murant, A.F., Abu Salih, H.S. & Goold, R.A. 1974. Viruses from broad bean in the Sudan. Ann. Rept. Scottish Hort. Res. Inst. 1973: 67.

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 notin embryonal axis (Makkouk *et al.*, 1987).

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Devergne, J.-C. & Cousin, R. 1966. Le virus de la mosaique de la fève (MF) et les symptomes d'ornementation sur graines. *Annls Epiphyties* **17** (No. H.S.):147-161.

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Tapio, E. 1970. Virus diseases of legumes in Finland and in the Scandinavian countries. *Annls Agric. Fenn.* **9**:1-97.

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

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Cockbain, A. J., Bowen, R. & Vorrai-Urai, S. 1976. Seed transmission of broad bean stain virus and Echtes, Ackerbohnenmosaik-Virus in field beans *(Vicia faba). Ann. appl. Biol.* **84**:321-332.

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Jones, A.T. 1980. Seed-borne viruses of *Vicia faba* and the possibility of producing seed free from broad bean stain virus and Echtes Ackerbohnen mosaik-Virus. pp. 319-333. In: Vicia faba, *feeding value, processing and viruses.* Ed. D.A. Bond. Martinus Nijhoff, the Hague.

Paul, H.L. & Quantz, L. 1959. Uber den Wechsel der Konzentration des Echten Ackerbohnenmosaik-Virus in Ackerbohnen. *Arch. Mikrobiol.* **32**:312-318.

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

Aboul Ata, A.E., Allen, D.J., Thottappilly, G. & Rossel, H.W. 1982. Variation in the rate of seed transmission of cowpea aphid-borne mosaic virus in cowpea. *Trop. Grain Leg. Bull.* **25**:2-7.

Behncken, G.M. & Maleevsky, L. 1977. Detection of cowpea aphid-borne mosaic virus in Queensland. *Austral. J. Exp. Agric. Anim. Husb.* **17**:647-678.

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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.

Hino, T. 1962. Studies on the asparagus-bean mosaic virus. *Ann. Phytopath. Soc. Japan* **25**:178-186.

Kaiser, W.J. & Mossahebi, G.H. 1975. Studies with cowpea aphid-borne mosaic virus and its effect on cowpea in Iran. *FAO Plant Prot. Bull.* **23**:33-39.

Lovisolo, O. & Conti, M. 1966. Identification of an aphid-transmitted cowpea mosaic virus. *Neth. J. Plant Path.* **72**:265-269.

Purcifull, D. & Gonsalves, D. 1985. Blackeye cowpea mosaic virus. AAB Descriptions of Plant Viruses No. 305. Association of Applied Biologists, Wellesbourne.

Tsuchizaki, T., Yora, K. & Asuyama, H. 1970a. The viruses causing mosaic of cowpea and Azuki bean, and their transmissibility through seeds. *Ann. Phytopath. Soc. Japan* **36**:112-120.

Tsuchizaki, T., Yora, K. & Asuyama, H. 1970b. Seed transmission of viruses in cowpea and Azuki bean plants. *Ann. Phytopath. Soc. Japan* **36**:237-242.

11. Cowpea mild mottle virus

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

Most range

Reported from cowpea (Brunt & Menten, 1973), bambara groundnut *(Vigna subterranea),* soybean, winged bean *(Psophocarpus tetragonolobus)* (Fauquet *et al.,* 1979; Thouvenel *et al.,* 1982), groundnut (Iizuka *et al.,* 1984), mungbean (Mink & Keswani, 1987), and some leguminous weed species (Anno-Nyako, 1984). Commonly found in common bean, and lima bean *(Phaseolus lunatus)* in Nigeria, in which it causes prominent disease symptoms (Rossel, unpublished). Also reported from tomato (Brunt & Phillips, 1981). Experimentally transmissible to other legume crop species and some test plant species including *Nicotiana clevelandii* and *N. megalosiphon* (Anno-Nyako, 1984).

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

Anno-Nyako, F.O. 1984. Identification and partial characterization of a mild mottle disease in soybean *(Glycine max* (L) Merril) in Nigeria. Ph.D. Thesis, University of Science and Technology, Kumasi, Ghana.

Brunt, A.A. & Kenten, R.H. 1973. Cowpea mild mottle, a newly recognized virus infecting cowpeas *(Vigna unguiculata)* in Ghana. *Ann. appl. Biol.* **74**:67-74.

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Brunt, A.A. & Phillips, S. 1981. 'Fuzzy vein', a disease of tomato *(Lycopersicon esculentum)* in western Nigeria induced by cowpea mild mottle virus. *Trop. Agric.* (Trinidad) **58**:177-180.

Fauquet, C., Lamy, D. & Thouvenel, J.C. 1979. Viral diseases of winged bean in the Ivory Coast. *FAO Plant Prot. Bull.* **27**:81-87.

Iizuka, N., Rajeshwari, R., Reddy, D.V.R., Goto, F., Munyappa, V., Bharathan, N. & Ghanekar, A.M. 1984. Natural occurrence of a strain of cowpea mild mottle virus on groundnut *(Arachis hypogaea)* in India. *Phytopath. Z.* **109**:245-253.

Mink, G.I. & Keswani, C.L. 1987. First report of cowpea mild mottle virus on bean and mung bean in Tanzania. *Plant Dis.* **71**:557.

Thouvenel, J.C., Montsarrat, A. & Fauquet, C. 1982. Isolation of cowpea mild mottle virus from diseased soybeans in Ivory Coast. *Plant Dis.* **66**:336-337.

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 l-2 to more than 8 days (Van Kammen & De Jager, 1978)., Suspected seed transmission (l-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

Bock, K.R. 1971. Notes on East African plant virus diseases. I. Cowpea mosaic virus. *E.A. Agric. Forestry* 1. **37**:60-62.

Chant, S.R. 1959. Viruses of cowpea, *Vigna unguiculata* (L.) Walp. in Nigeria. *Ann. appl. Biol.* **47**:565-572.

Gilmer, R.M., Whitney, W.K. & Williams, R.J. 1973. Epidemiology and control of cowpea mosaic virus in Western Nigeria. Proc. Ist IITA Grain Leg. Impr. Workshop: 269.

Kammen, A. van & Jager, C.P. de. 1978. Cowpea mosaic virus. CMI/AAB Descriptions of Plant Viruses No. 197. Commonwealth Agricultural Bureaux, Slough.

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Patel, P.N. & Kuwite, C. 1982. Prevalence of cowpea aphid-borne mosaic virus and two strains of cowpea mosaic virus in Tanzania. *Indian Phytopath.* **35**:467-472.

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Thottappilly, G. & Rossel, H.W. 1987. Seed transmission of cowpea (yellow) mosaic virus unlikely in cowpea. *Trop. Grain Leg. Bull.* **34**:27-28.

Whitney, W.K. & Gilmer, R.M. 1974. Insect vectors of cowpea mosaic virus in Nigeria. *Ann. appl. Biol.* **77**:12-21.

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

Allen, D.J. 1980. Identification of resistance to cowpea mottle virus. *Trop. Agric.* (Trinidad) **57**:325-332.

Allen, D.J., Thottappilly, G. & Rossel, H.W. 1982. Cowpea mottle virus: field resistance and seed transmission in virus-tolerant cowpea. *Ann. appl. Biol.* **100**:331-336.

Boswell, K.F. & Gibbs, A.J. (eds.). 1983. *Viruses of legumes* 1983. Descriptions and keys from VIDE. Austral. Nat1 Univ., Canberra.

Robertson, D.G. 1966. Seed-borne viruses of cowpea in Nigeria. B.Sc. Thesis, University of Oxford, Oxford.

Rossel, H.W. 1977. Preliminary investigations on the identity and ecology of legume virus diseases in northern Nigeria. *Trop. Grain Leg. Bull.* **8**:41-46.

Shoyinka, S., Bozarth, R.F., Reese, J. & Rossel, H.W. 1978. Cowpea mottlevirus, a seed-borne virus with distinctive properties infecting cowpeas in Nigeria. *Phytopathology* **68**:693-699.

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

Phatak, H.C. 1974. Seed-borne plant viruses - identification and diagnosis in seed health testing. *Seed Sci. & Technol.* **2**:3-155.

Phatak, H.C., Diaz-Ruiz, J.R. & Hull, R. 1976. Cowpea ringspot virus: a seed-transmitted cucumovirus. *Phytopath. Z.* **87**:132-142.

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

Agrawal, H.O. 1964. Identification of cowpea mosaic virus isolates. *Meded. Landb.Hogesch. Wageningen* **64**(5):53 pp.

Dale, W.T. 1949. Observations on a virus disease of cowpea in Trinidad. *Ann. appl. Biol.* **36**:327-333.

Haque, S.Q. & Persad, G.C. 1975. Some observations on the seed-transmission of beetle-transmitted cowpea mosaic virus. pp. 119-121. In: *Tropical diseases of legumes.* Eds. J. Bird & K. Maramorosch, Academic Press, New York.

Hoof, H.A. van. 1963. Transmission of cowpea mosaic virus in Surinam. *Surin. Landb.* **11**:131-137.

Jager, C.P. de. 1979. Cowpea severe mosaic virus. CMI/AAB Descriptions of Plant Viruses No. 209. Commonwealth Agricultural Bureaux, Slough.

Lin, M.T., Anjos, J.R.N. & Rios, G.P. 1982. Cowpea severe mosiac virus in five legumes in central Brazil. *Plant Dis.* **66**:67-70.

Shepherd, R.J. 1964. Properties of a mosaic virus of cowpea and its relationship to the bean pod mottle virus. *Phytopathology* **54**:466-473.

Swaans, H., & Kammen, A. van. 1973. Reconsideration of the distinction between the severe and yellow strains of cowpea mosaic virus. *Neth. J. Plant Path.* **79**:257-265.

Thongmeearkom, P. & Goodman, R.M. 1976. A severe disease of soybean caused by an isolate of cowpea mosaic virus. *Proc. Am. Phytopath.* Soc. **3**:209-210 (Abstr.).

16. Cryptic (or temperate) viruses

Cryptovirus group; spherical particles c. 30 nm in diameter with segmented dsRNA of about 4 x 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

Abou-Elnasr, M.A., Jones, A.T. & Mayo, M.A. 1985. Detection of dsRNA in particles of Vicia cryptic virus and in *Vicia faba* tissues and protoplasts. *J. gen. Virol.* **66**:2453-2460.

Boccardo, G., Lisa, V. & Milne, R.G. 1983: Cryptic viruses in plants. pp. 425-430. In: *Double-stranded RNA viruses.* Eds. R.W. Compans & D.H.L. Bishop. Elsevier, Amsterdam.

Boccardo, G., Milne, R.G., Luisoni, E., Lisa, V. & Accotto, G.P. 1985. Three seedbome cryptic viruses containing double-stranded RNA isolated from white clover. *Virology* **147**:29-40.

Kenten, R.H., Cockbain, A.J. & Woods, R.D. 1980. Vicia cryptic virus. Rothamsted Exp. Stn. Ann. Rep. 1979: 176.

Natsuaki, T., Natsuaki, K.T., Okuda, S., Teranaka, M., Milne, R.G., Boccardo, G. & Luisoni, E. 1986. Relationships between the cryptic and temperate viruses of alfalfa, beet and white clover. *Intervirology* **25**:69-75.

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

BOS, L. & Maat, D.Z. 1974. A strain of cucumber mosaic virus, seed-transmitted in beans. *Neth. J. Plant Path.* **80**:113-123.

Chang, C.A. 1983. Rugose mosaic of asparagus bean. caused by dual infection with cucumber mosaic virus and blackeye cowpea mosaic virus. *Plant Prot. Bull.* (Taiwan) **25**:177-190.

Francki, R.I.B., Mossop, D.W. and Hatta, T. 1979. Cucumber mosaic virus. CMI/AAB Descriptions of Plant Viruses No. 213. Commonwealth Agricultural Bureaux, Slough.

Golebniak, B. 1979. Transmission of cucumber mosaic virus by seeds of blue and yellow lupins. *Zesz. Probl. Post. Nauk Roln.* **226**:99-102.

Green, SK. 1985. Cucumber mosaic virus: host range, seed transmission, and sources of resistance. Progress Report AVRDC 1985:171-l74.

Iwaki, M. 1978. Seed transmission of a cucumber mosaic virus in mungbean *(Vigna radiata). Ann. Phytopath. Soc. Japan* **44**:337-339.

Jones, R.A.C. 1988. Seed-borne cucumber mosaic virus infection of narrow-leafed lupin *(Lupinus angustifolius)* in Western Australia. *Ann. appl. Biol.* **113**:507-518.

Meiners, JP., Waterworth, H.E., Smith, F.F., Alconero, R. & Lawson, R.H. 1977. A seed-transmitted strain of cucumber mosaic virus isolated from bean. *J. Agric. Univ. Puerto Rico* **61**:137-147.

Phatak, .H.C 1974. Seed-borne plant viruses. Identification and diagnosis in seed health testing. *Seed Sci. & Technol.* **2**:3-155.

Pio-Riberio, G., Wyatt, S.D. & Kuhn, C.W. 1978. Cowpea stunt: A disease caused by a synergistic interaction of two viruses. *Phytopathology* **68**:1260-1265.

Prakash, J. & Joshi, R.D. 1980. Some aspects of seed transmission of cowpea banding mosaic virus in cowpea. *Seed Sci. & Technol.* **8**:393-399.

Purivirojkul, W., Silleyos, P., Hsu, C.H., Paehlman, J.H. & Sehgal, O.P. 1978. Natural infection of mung bean *(Vigna radiata)* with cucumber mosaic virus. *Plant Dis. Reptr* **62**:530-534.

Xu, Z. & Barnett, O.W. 1984. Identification of a cucumber mosaic virus strain from naturally infected peanuts in China. *Plant Dis.* **68**:386-389.

18. Guar symptomless virus

Potyvirus group; flexuous particles c. 760 nm; transmitted in sap (Hansen & Lesemann, 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

Behncken, G.M. 1983. Guar symptomless virus. pp. 59-60. In: *Viruses of legumes* 1983. Eds. K.F. Boswell & A.J. Gibbs. Descriptions and keys from VIDE. Austral. Natl Univ., Canberra.

Hansen, A.J. & Lesemann, D.E. 1978. Occurrence and characteristics of a seed-transmitted potyvirus from Indian, African and North American guar. *Phytopathology* **68**:841-846.

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).

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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.

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21. Pea early-browning virus

Tobravirus 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

Causes disease in pea, common bean, faba bean (Bos & van der Want, 1963; Gibbs & Harrison, 1964; Lockhart & Fischer, ‘1976: Gerhardson & Ryden, 1979; Fiedorow, 1980, l983) and yellow lupin (Pospieszny & Frencel, 1985), and infects symptomlessly other legumes, including lucerne, and some non-legumes (Bos & van der Want, 1963).

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 *(Trichodurus* spp.). Above-ground spread is by seed. Rate of transmission in pea is 1 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).

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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).

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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

Infects 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.

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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).

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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).

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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

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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, often 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 (l-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).

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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).

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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.

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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.

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Graddon, D.J. & Randles, J.W. 1986. Single antibody dot immunoassay - a simple technique for rapid detection of a plant virus. *J. Virol. Methods* **13**:63-69.

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 1eaves.

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 (Capoor, 1962; Nagaich & Vashisth, 1963; Capoor *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'.

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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, al though 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).

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33. Tobacco streak virus

Ilarvirus 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 appearence.

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

Costa, A.S. & Carvalho, A.M.B. 1961. Studies on Brazilian tobacco streak. *Phytopath. Z.* **42**:113-138.

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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 *Phaseulus vulgaris);* gel-diffusion serology.

References

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35. Urdbean leaf crinkle virus

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

Host range

Urdbean *(Phaseolus (Vigna) mungo)* cowpea, mungbean *(V. radiata),* pigeon pea *(Cajanus cajan)* and tepary bean *(Phaseolus (Vigna) aconitifolius)* (Kolte & Nene, 1975; Beniwal, 1983).

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.*

References

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Table 1. Naturally seed-transmitted viruses occurring in different legumes.

Host	Virus	% transmission	Reference	Geographical distribution
Arachis hypogaea	Cucumber mosaic	0-2	82	China
	Peanut clump	6-14	76	Burkina Faso, Côte d'Ivoire, India, Niger, Senegal
	Peanut mottle	0-8.5	2,13	Probably worldwide
	Peanut stripe	0.1-10	25	China, India, Indonesia, Japan, Malaysia, Myanmar (Burma), Philippines, Thailand, USA, Vietnam
	Peanut stunt	0.1	78	Europe, Africa, Asia, North America, Japan
Crotalaria juncea	Sunn-hemp mosaic	10-20	80	India
Cyamopsis tetragonoloba	Guar symptomless	12-70	39	Australia, India, Pakistan, USA
Desmodium canum	Desmodium mosaic	8	27	USA (Florida)
Glycine max	Bean pod mottle	0.08	56	USA (Nebraska)
	Cucumber mosaic	30-100	74	Indonesia, Japan, USA, USSR
	Soybean mosaic	0.1-30	16,36	Worldwide
	Soybean stunt	0-50	54	Japan
	Tobacco ringspot	0-100	6	North America
	Tobacco streak	0-90	31,79	Argentina, Brazil, USA
Lens culinaris	Broad bean stain	14	57	Syria
	Peaseedborne mosaic	5-44	37,38	USA
Lupinus albus	Cucumber mosaic	**	35	Europe
	Peanut mottle	0.4	24	USA (Georgia)
Lupinus angustifolius	Cucumber mosaic	3-34	4,47	Australia
Lupinus luteus	Bean yellow mosaic	6	7,21	Worldwide

Table 1. Naturally seed-transmitted viruses occurring in different legumes (cont'd).

Host	Virus	% transmission	Reference	Geographical distribution
Lupinus luteus	Cucumber mosaic	21	84	Europe
Macroptilium lathyroides	Bean common mosaic	5-33	68	Guyana, Hawaii, Philippines, Suriname
Medicagopolymorpha	Alfalfa mosaic	0.2-49	46	Australia
Medicago sativa	Alfalfa mosaic	l-30	66	Worldwide
	Lucerne Australian latent	0-8	10	Australia, New Zealand
Medicago truncatula	Alfalfa mosaic	2	46	Australia
Melilotus albus	Lucerne transient streak	2.5	10,65	Australia, Canada, New Zealand
	Tobacco streak	0-3	51	USA
Phaseolus acutifolius var. *latifolius*	Bean common mosaic	7-22	69	USA
Phaseolus vulgaris	Bean common mosaic	0-83	60,62	Worldwide
	Cucumber mosaic	0-7	14,59	Worldwide
	Peanut mottle	0-1	8	Australia (Queensland)
	Southern bean mosaic	l-30	43,61	Africa, Americas, India
	Tobacco streak	0-27	75	USA
	Tomato aspermy	19	81	China
Pisum sativum	Bean yellow mosaic	5	26	Worldwide
	Peas eedborne mosaic	10-100	53	Worldwide
	Pea early-browning	1-37	15,41	Europe, Morocco
	Pea mild mosaic	15	19	New Zealand
Trifolium subterraneum	Subterranean clover mottle	3	30	Australia
Viciafaba	Bean yellow mosaic	0.1-2.4	48,63	Iran, Sudan

Table 1. Naturally seed-transmitted viruses occurring in different legumes (cont'd).

Host	Virus	% transmission	Reference	Geographical distribution
Viciafaba	Broad beanmottle	1-2	58	North Africa, Portugal, UK, Syria, Sudan
	Broad bean stain	l-10	32,44,45	Australia*, China*, Europe, North Africa, Sudan
	Broad bean true mosaic	1-17	11,12,20,44	Australia*, Europe, northwest Africa
	Pea seed bome mosaic	**	64	Europe
	Pea early-browning	l-10	28,29	Europe
Vigna catjang	Sunn-hemp mosaic	17	18	India
Vigna mungo	Bean common mosaic	2-10	3	India
	Blackgram mottle	8	70	India, Thailand
	Urdbean leaf crinkle	18	9	India
Vigna radiata	Bean common mosaic	8-32	49	Iran
	Cucumber mosaic	10	42	Japan
Vigna sesquipedalis	Cowpea severe mosaic	8	22	South America, southern USA
Vigna unguiculata	Blackeye cowpea mosaic	30	83	Worldwide
	Cowpea aphid-borne mosaic	7-18	1,50	Worldwide
	Cowpea mild mottle	90	17,77	Worldwide
	Cowpea mosaic	1-5	33	Cuba, Kenya, Nigeria, Suriname, USA
	Cowpea mottle	0.2-10	5,73	Nigeria
	Cowpea ringspot	10-30	67	Iran
	Cowpea severe mosaic	l-10	22,40,71	Americas, Puerto Rico, Trinidad
	Cucumber mosaic	15-20	67	Worldwide
	Peanut mottle	< l	23	USA (Georgia)
	Southern bean mosaic	l-40	34,55,72	Africa, Americas
	Sunn-hemp mosaic	4-20	52	Africa, India, US A
	Urdbean leaf crinkle	6-15	9	India

* Detected in small plantings and eradicated

** Data on rate of seed transmission not available

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Table 2. Legume hosts in which natural seed-transmission has been reported.

Virus	Host	% transmission	Reference	Geographical distribution
Alfalfa mosaic	*Medicago polymorpha*	0.2-49	46	Australia
	M. sativa	1-30	66	Worldwide
	M. truncatula	2	46	Australia
Bean common mosaic	*Macroptilium lathyroides*	5-33	68	Guyana, Hawaii, Philippines, Suriname
	Phaseolus acutifolius var. *latifolius*	7-22	69	USA
	P. vulgaris	0-83	60,62	Worldwide
	Vigna mungo	2-10	3	India
	V. radiata	8-32	49	Iran
Bean pod mottle	*Glycine max*	0.08	56	USA (Nebraska)
Bean yellow mosaic	*Lupinus luteus*	6-14	7-21	Worldwide
	Pisum sativum	5	26	Worldwide
	Vicia faba	0.1-2.4	48,63	Iran, Sudan
Blackeye cowpea mosaic	*Vigna unguiculata*	30	83	Worldwide
Blackgram mottle	*Vigna mungo*	8	70	India, Thailand
Broad bean mottle	*Vicia faba*	1-2	58	North Africa, Portugal, UK, Syria, Sudan
Broad bean stain	*Vicia faba*	1-10	32,44,45	Australia*, China*, Europe, North Africa, Sudan
	Lens culinaris	14	57	Syria
Broad bean true mosaic	*Vicia faba*	1-17	11,12,20,44	Australia*, Europe, northwest Africa
Cowpea aphid-borne mosaic	*Vigna unguiculata*	0-40	1,50	Worldwide

Table 2. Legume hosts in which natural seed-transmission has been reported (cont'd).

Virus	Host	% transmission	Reference	Geographical distribution
Cowpea mild mottle	*Vigna unguiculata*	0-90	17,77	Worldwide
Cowpea mosaic	*Vigna unguiculata*	1-5	33	Cuba, Kenya, Nigeria, Suriname, USA
Cowpea mottle	*Vigna unguiculata*	0.2-10	5,73	Nigeria
Cowpea ringspot	*Vigna unguiculata*	10-30	67	Iran
Cowpea severe mosaic	*Vigna unguiculata*	l-10	40,71	Americas, Puerto Rico, Trinidad
	V. sesquipedalis	8	22	South America, southern USA
Cucumber mosaic	*Arachis hypogaea*	0-2	82	China
	Glycine max	30- 100	74	Indonesia, Japan, USA, USSR
	Lupinus albus	**	35	Europe
	L. angustifolius	3-34	4,47	Australia
	L. luteus	21	84	Europe
	Phaseolus vulgaris	0-7	14,59	Worldwide
	Vigna radiata	10	42	Japan
	V. unguiculata	15-20	67	Worldwide
Desmodium mosaic	*Desmodium canum*	8	27	USA (Florida)
Guar symptomless	*Cyamopsis tetragonoloba*	12-70	39	Australia, India, Pakistan, USA
Lucerne Australian latent	*Medicago sativa*	0-8	10	Australia, New Zealand
Lucerne transient streak	*Melilotus albus*	2.5	10,65	Australia, Canada, New Zealand
Peaseedborne mosaic	*Lens culinaris*	5-44	37,38	USA
	Pisum sativum	10-100	53	Worldwide
	Viciafaba	**	64	Europe

Table 2. Legume hosts in which natural seed-transmission has been reported (cont'd),

Virus	Host	% transmission	Reference	Geographical distribution
Pea early-browning	*Pisum sativum*	1-37	15,41	Europe, Morocco
	Viciafaba	l-10	28, 29	Europe
Pea mild mosaic	*Pisum sativum*	15	19	New Zealand
Peanut clump	*Arachis hypogaea*	6-14	76	Burkina Faso, Côte d'Ivoire, India, Niger, Senegal
Peanut mottle	*Arachis hypogaea*	0-20	2-13	Probably worldwide
	Lupinus albus	0.4	24	USA (Georgia)
	Phaseolus vulgaris	0-1	8	Australia (Queensland)
	Vigna unguiculata	< l	23	USA (Georgia)
Peanut stripe	*Arachis hypogaea*	0.1-10	25	China, India, Indonesia, Japan, Malaysia, Myanmar (Burma), Philippines, Thailand, USA, Vietnam
Peanut stunt	*Arachis hypogaea*	0.1	78	Europe, Africa, Asia, North America, Japan
Southern bean mosaic	*Phaseolus vulgaris*	l-30	43,61	Africa, Americas, India
	Vigna unguiculata	l-40	34,55,72	Africa, Americas
Soybean mosaic	*Glycine max*	0.1-30	16,36	Worldwide
Soybean stunt	*Glycine max*	0-50	54	Japan
Subterranean clover mottle	*Trifolium subterraneum*	3	30	Australia
Sunn-hemp mosaic	*Crotalaria juncea*	10-20	80	India
	Vigna catjang	17	18	India
	V. unguiculata	4-20	52	Africa, Australia, India, USA

Table 2. Legume hosts in which natural seed-transmission has been reported (cont'd).

Virus	Host	% transmission	Reference	Geographical distribution
Tobacco ringspot	*Glycine max*	0-100	6	Egypt, India, North America, Sri Lanka, Turkey
Tobacco streak	*Glycine max*	0-90	31-79	Argentina, Brazil, USA
	Melilotus albus	0-3	51	USA
	Phaseolus vulgaris	0-27	75	USA
Tomato aspermy	*Phaseolus vulgaris*	19	81	China
Urdbean leaf crinkle	*Vigna mungo*	18	9	India
	V. unguiculata	6-15	9	India

* Detected in small plantings and eradicated

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Bacterial diseases

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 theneither 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 (l-5 mins) or dips in sodium hypochlorite (l-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.

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

Bacterium		Principal leguminous host
Clavibacter michiganense* subsp. *insidiosum*		alfalfa / lucerne
Curtobacterium flaccumfasciens* pv. *flaccumfasciens*		bean
Pseudomonas solanacearum		groundnut
Pseudomonas syringae	pv. *glycinea*	soybean
	pv. *phaseolicola*	common bean
	pv. *pisi*	pea
	pv. *syringae*	-
	pv. *tabaci*	soybean
Xanthomonas campestris	pv. *alfalfae*	alfalfa / lucerne
	pv. *cajani*	pigeonpea
	pv. *cassiae*	chickpea
	pv. *cyamopsidis*	clusterbean
	pv. *glycinea*	soybean
	pv. *phaseoli*	common bean
	pv. *pisi*	pea
	pv. *vignaeradiatae*	mungbean
	pv. *vignicola*	cowpea

* 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).

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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
Seed transmitted in *Glycine max* (Coerper, 1919; Nicholson & Sinclair, 1971; Leben, 1975).

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3. Bacterial brown spot

Cause
Pseudomonas syringae pv. *syringae* van Hall.

Symptoms
Brown spots on 1eaves and pods, shrivelled seeds.

Geographical distribution
Worldwide (Anonymous, 1988).

Host range
Many important legume and non-leguminous crops.

Biology and transmission
Seed transmission in *Phaseolus lunatus* (Thaung & Walker, 1957), *P. vulgaris* (Harrison & Freeman, 1965; Hoitink & Hagedorn, 1966; Hoitink *et al.*, 1968, *Vigna unguiculata* (Gardner & Kendrick, 1922; 1925; Hoffmaster, 1944).

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4. Bacterial wilt

Cause
Pseudomonas solanacearum (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
Widespread mainly within latitudes 40° N & S (Anonymous, 1977).

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).

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Palm, B.T. 1922. Aanteekeningen over slijmziekte in Arachis hypogaea (Katjang tanah). *Meded. Inst. voor Plantenziekten* (Buitenzorg) **52:** 1-41.

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

Anonymous. 1987. CMI distribution maps of plant diseases. No. 370 (edition 4). Commonwealth Agricultural Bureaux, Slough.

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Burkholder, W.H. 1926. A new bacterial disease of the bean. *Phytopathology* **16:** 915-927.

Dunleavy, J. 1962. Pigmentation of soybean seed coats associated with bacteria. *Phytopathology* **52:** 730. (Abstr.)

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Leonard, L.T. 1924. Effect of moisture on a seed-borne bean disease. *J. Agric. Res.* **28:** 489-497.

Schuster, M.L. & Coyne, D.P. 1974. Survival mechanisms of phytopathogenic bacteria. *Ann. Rev. Phytopath.* **12:** 199-221.

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
Anonymous. 1987. CMI distribution maps of plant diseases. No. 67 (edition 4). Commonwealth Agricultural Bureaux, Slough.

Cormack, M.W. 1961. Longevity of the bacterial wilt organism in alfalfa hay, pod debris and seed. *Phytopathology* **51:** 260-261.

Cormack, M.W. & Moffatt, J.E. 1955. Seed transmission of bacterial wilt of alfalfa. *Proc. Can. Phytopath. Soc.* **23:** 15. (Abstr.)

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Hayward, A.C. & Waterson, J.M. 1964. *Corynebacterium insidiosum* . CMI Description of Pathogenic Fungi and Bacteria. No. 13. Commonwealth Agricultural Bureaux, Slough.

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

Seed transmitted in *Phaseolus vulgaris* (Zaumeyer, 1929; Burkholder, 1930; Wallen & Sutton, 1965; Saettler & Perry, 1972). A common variant, formerly referred to as var. *fuscans*, produces a brown diffusible pigment in agar culture.

References

Anonymous. 1971. CMI distribution maps of plant diseases. No. 401 (edition 2). Commonwealth Agricultural Bureaux, Slough.

Burkholder, W.H. 1930. The bacterial disease of the bean. A comparative study. New York (Cornell) Agr. Exp. Sta., Mem. 127. 93p.

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Saettler, A.W. & Perry, S.K. 1972. Seed transmitted bacterial diseases in Michigan navy (pea) beans, *Phaseolus vulgaris. Plant Dis. Reptr* **56**:378-381.

Wallen, V.R. & Sutton, M.D. 1965. *Xanthomonas phaseoli* var *fuscans* (Burkh.) Starr and Burkh. on field bean in Ontario. Can. J. Bot. **43**:437-446.

Zaumeyer, W.J. 1929. Seed infection by *Bacterium phaseoli. Phytopathology* **19**:96.

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

Cajanus cajan, Lablab purpureus, Macroptilium spp., *Phaseolus coccineus, P. lunatus, P. vulgaris, Pueraria* spp., *Vigna angularis, V. radiata, Neonotonia wightii.*

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

Anonymous. 1973. CMI distribution maps of plant diseases. No. 85 (edition 4). Commonwealth Agricultural Bureaux, Slough.

Burkholder, W.H. 1926. A new bacterial disease of the bean. *Phytopathology* **16:** 915-927.

Grogan, R.G. & Kimble, K.A. 1967. The role of seed contamination in the transmission of *Pseudomonas phaseolicola* in *Phaseolus vulgaris. Phytopathology* **57:** 28-32.

Hayward, A.C. & Waterson, J.M. 1965. *Pseudomonas solanacearum.* CMI Description of Pathogenic Fungi and Bacteria. No. 45. Commonwealth Agricultural Bureaux, Slough.

Katznelson, H., Sutton, M.D. & Bayley, S.T. 1954. The use of bacteriophage of *Xanthomonas phaseoli* in detecting infection in beans, with observations on its growth and morphology. *Can. J. Microbiol.* **1:** 22-29.

Taylor, J.D. 1970. The quantitative estimation of the infection of bean seed with *Pseudomonas phaseolicola* (Burkh.) Dowson. *Ann appl. Biol.* **66:** 29-36.

Taylor, J.D., Teverson, D.M. & Davis, J.H.C. 1987. Halo-blight of *Phaseolus* bean. Report of the National Vegetable Research station for 1986, pp.63-64. National Vegetable Research Station, Wellesbourne.

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

Anonymous. 1986a. CMI distribution maps of plant diseases. No 328 (edition 3). Commonwealth Agricultural Bureaux, Slough.

Anonymous. 1986b. *Phaeoisariopsis griseola.* CMI Description of Pathogenic Fungi and Bacteria. No. 874. Commonwealth Agricultural Bureaux, Slough.

Bose, S.K. & Sindhan, G.S. 1972. Leaf spot of French beans caused by *Isariopsis griseola* Sacc. and its control. *Progress. Hort.* **4:** 69-75.

Orogoco-Sarria, S.H. & Cordona Alvarez, C. 1959. Evidence of seed transmission of angular leaf spot of bean. *Phytopathology* **49:** 159.

Sohi, H.S. & Sharma, R.D. 1974. Mode of survival of *Isariopsis griseola* Sacc., the causal agent of angular leaf spot of beans. *Ind. J. Hort.* **31:** 110-113.

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

Haware, M.P., Nene, Y.L. & Mathur, S.B. 1986. Seed-borne diseases of chickpea. Technical Bulletin No. 1. Danish Government Institute of Seed Pathology for Developing Countries, Copenhagen.

Maden, S., Singh, D., Mathur, S.B. & Neergaard, P. 1975. Detection and location of seed-borne inoculum of *Ascochyta rabiei* and its transmission in chickpea *(Cicer arietinum). Seed Sci. & Technol.* **3:** 667-681.

Mathur, S.B. 1981. ISTA Handbook on Seed Health Testing, Section 2, Working Sheet No. 38. International Seed Testing Association, Zurich.

Reddy, M.V. 1980. Calixin M - an effective fungicide for eradication of *Ascohyta rabiei* in chickpea seeds. *International Chickpea Newsletter* **3:** 12.

Vir, S. & Grewal, J.S. 1975. Physiologic specialization in *Ascochyta rabiei*, the causal organism of gram blight. *Indian Phytopath.* **27:** 335-360.

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

Anselme, C. & Champion, R. 1981. ISTA Handbook on Seed Health Testing. Section 2, Working Sheet No. 45. International Seed Testing Association, Zurich.

Cruickshank, I.A.M. 1966. Strains of *Colletotrichum lindemuthianum* (Sacc. & Magn.) in Eastern Australia. *J. Aust. Inst. Agric. Sci.* **32:** 134-135.

Kummer, H. & Schmidt, B. 1961. Health examination and assessment of anthracnosed bean and pea seed. *Zbl. Bakt.* **114:** 616-630.

Petrov, B. 1972. Anthracnose of bean. *Rastit. Zasht.* **20:** 24-26.

Sindhan, G.S. & Bose, S.K. 1981. Evaluation of fungicides against anthracnose of French bean caused by *Colletotrichum, lindemuthianum. Indian Phytopath.* **34:** 325-329.

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

Sinclair, J.B. & Backman, P.A. (eds.). 1989. *Compendium of soybean disease.* American Phytopathological Society, St. Paul.

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

Jackson, C.R. & Bell, D.K. 1969. Diseases of peanut (groundnut) caused by fungi. *University of Georgia Agric. Exp. Stn Res. Bull.* **56:** 91-100.

Lal. S.P., & Mathur, S.B. 1967. Studies on seed-borne fungi of groundnut. I. A new method for detecting fungi in small samples of seed, with special reference to *Macrophomina phaseoli* (Maubl.) Ashby (syn. *Sclerotium bataticola* Taub.). II. Laboratory assay of fungicides for controlling seed-borne infections. *Proc. Int. Seed Test. Ass.* **37:** 655-666.

McDonald, D. & Mehan, V.K. 1984. Charcoal rot. pp. 27-28. In: *Compendium of peanut diseases.* Eds. D.M. Porter, D.H. Smith and R. Rodriguez-Kabana. American Phytopathological Society, St. Paul.

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

Widespread (Anonymous, 1979).

Alternative hosts

Not known.

Biology and transmission

Systemically transmitted to seedlings (Novakova & Pfeiferova, 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.

References

Anonymous. 1979. CMI distribution maps of plant diseases. No. 268 (edition 3). Commonwealth Agricultural Bureaux, Slough.

Hansen, H. J. & Mathur, S.B. 1987. ISTA Handbook on Seed Health Testing, Section 2, Working Sheet No. 64. International Seed Testing Association, Zurich.

Novakova, J. & Pfeiferova, J. 1964. A contribution to the study of *Peronospora manshurica* on soybean in Czechoslovakia. *Ceská Mykol.* **l8:** 42-47.

Sinclair, J.B. & Backman, P.A. (eds.). 1989. *Compendium of soybean diseases.* American Phytopathological Society, St. Paul.

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.

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

Anonymous. 1985. CMI distribution maps of plant diseases. No. 166 (edition 5). Commonwealth Agricultural Bureaux, Slough.

Jackson, C.R. & Bell, D.K. 1969. Diseases of peanut (groundnut) caused by fungi. *University of Georgia Agric. Exp. Stn Res. Bull.* **56:** 7-15.

McDonald, D., Subrahmanyam, P., Gibbons, R.W. & Smith, D.H. 1985. Early and late leaf spots of groundnut. ICRISAT Information Bulletin No. 21. International Crops Research Institute for the Semi-Arid Tropics, Patancheru.

Porter, D.M., Smith, D.H. & Rodriguez-Kabana, R. 1982. Peanut diseases. pp. 326-410. In: *Peanut science and technology*, American Peanut Research and Education Society, Yoakum.

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

Anonymous. 1985. CMI distribution maps of plant diseases. No. 160 (edition 6). Commonwealth Agricultural Bureaux, Slough.

Peregrine, W.T.H. 1971. Groundnut rust *(Puccinia arachidis)* in Brunei. PANS **17**:318-319.

Subrahmanyam, P. & McDonald, D. 1982. Groundnut rust - its survival and carry-over in India. Proceedings of the Indian Academy of Sciences. *Plant Science* **91**:93-100.

Subrahmanyam, P., McDonald, D. & Hammons, R.O. 1984. Groundnut rust. pp. 7-9. In: *Compendium of peanut diseases.* Eds. D.M. Porter, D.H. Smith & R. Rodriguez-Kabana. American Phytopathological Society, St. Paul.

Varma, B.K. & McDonald, D. 1984. Groundnut rust disease and plant quarantine. pp. 55-58. In: Proceedings of discussion group meeting, 24-28 September 1984. International Crops Research Institute for the Semi-Arid Tropics, Patancheru.

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

Giorda, L.M. 1984. Scab. p. 12. In: *Compendium of peanut diseases.* Eds. D.M. Porter, D.H. Smith & R. Rodriguez-Kabana. American Phytopathological Society, St. Paul.

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

Anonymous. 1987. CMI distribution maps of plant diseases. No. 152 (edition 5). Commonwealth Agricultural Bureaux, Slough.

Jackson, C.R. & Bell, D.K. 1969. Diseases of peanut (groundnut) caused by fungi. *University of Georgia Agric. Exp. Stn Res. Bull.* **56:** 7-15.

McDonald, D., Subrahmanyam, P., Gibbons, R.W. & Smith, D.H. 1985. Early and late leaf spots of groundnut. ICRISAT Information Bulletin No. 21. International Crops Research Institute for the Semi-Arid Tropics, Patancheru.

Porter, D.M., Smith, D.H. & Rodriguez-Kabana, R. 1982. Peanut diseases. pp. 326-410. In: *Peanut science and technology,* American Peanut Research and Education Society, Yoakum.

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
Leptospaerulina 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
Jackson, C.R. & Bell, D.K. 1969. Diseases of peanut (groundnut) caused by fungi. *University of Georgia Agric. Exp. Stn Res. Bull.* **56:** 37-43.

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 recommended by Papavizas & Lewis (1976).

References

Keeling, B.L. 1980. Research on *Phytophthora* root and stem rot: Isolation, testing procedures, and seven new physiological races. pp. 367-370. In: World Soybean Research Conference II: Proceedings, Ed. F.T. Corbin. Westview Press, Boulder.

Keeling, B.L. 1982. Four new physiological races of *Phytophthora megasperma* f. sp. *glycinea. Plant Dis.* **66:** 334-335.

Papavizas, G.C. & Lewis, J.A. 1976. Acetone infusion of pyroxychlor into soybean seed for the control of *Phytophthora megasperma* var. *sojae. Plant Dis. Reptr* **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

Haware, M.P., Nene, Y.L. & Rajeshwari, R. 1978. Eradication of *Fusarium oxysporum* f.sp. *ciceri* transmitted in chickpea seed. *Phytopathology* **68:** 1364-1367.

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:l) should eradicate the internal seed-borne *F. udum.*

Appendix : Accepted latin names and vernacular names of some legumes*

Latin name	English	French	Spanish	German	Other
Arachis hypogaea	groundnut	arachide	mani	gemeine Erdnuss	kacang tanah
Cajanus cajan	pigeonpea	pois d'Angole	guisante de paloma	Straucherbse	red gram
Canavalia ensiformis	jack bean	haricot sabre	haba de burro		fève Jacques
Canavalia gladiata	sword bean	pois sabre	haba de burro		kacang parang
Cicer arietinum	chickpea	pois chiche	garbanzo	Kichererbse	chana, Bengal gram
Cyamopsis tetragonoloba	cluster bean	cyamopse á 4 ailes			guar, aconite bean
Glycine max	soyabean	soja	soja	Sojabohne	soybean
Lablab purpureus	lablab	dolique d'Egypte		Helmbohne	hyacinth bean
Lathyrus sativus	grass pea	gesse commune	almorta	Saatplatterbse	khesari
Lens culinaris	lentils	lentille	lenteja	Linse	masur
Lupinus spp.	lupins	lupins	lupino	Lupinen	
Macrotyloma geocarpum	Kersting's groundnut	lentille de terre		Kandelabohne	ground bean kerstingiella
Macrotyloma uniflorum	horse gram	grain de cheval		Pferdekorn	kulthi, Madras gram
Mucuna pruriens	velvet bean	pois mascate	ojo de venado		
Pachyrhizus erosus	yam bean	dolique tubereux	jicama	knollige Bohne	Mexican yam bean
Pachyrhizus tuberosus	yam bean				
Phaseolus acutifolius	tepary bean	haricot riz	frijol trigo		Texan bean
Phaseolus lunatus	Lima bean	haricot de Lima	haba Lima	Limabohne	
Phaseolus vulgaris	common bean	haricot commun	frijol	Fisole	
Pisum sativum	pea	pois	guisante	Erbse	
Psophocarpus tetragonolobus	wing(ed) bean	pois ailé	sesquidilla	Goabohne	
Spenostylis stenocarpa	African yam bean				girigiri
Trigonella foenum-graecum	fenugreek				
Vicia faba	faba bean	fève	haba comun	Ackerbohne	
Vicia sativa	common vetch	vesce commune	veza	Futter-Wicke	
Vigna aconitifolia	moth bean	haricot papillon			math, phillipesa
Vigna mungo	black gram	ambérique	judia de urd	Urdbohne	urd
Vigna radiata	mung bean	haricot doré	judia de mungo	Mungobohne	green gram
Vigna subterranea	Bambara groundnut	voandzou			kacang Bogor
Vigna trilobata	pillipesara				jungli moth
Vigna umbellata	rice bean	haricot de riz	frijol arroz	Reisbohne	meth take-azuki
Vigna unguiculata	cowpea	pois vache	chicaro de vaca	Kuhbohne	kacang panjang

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

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