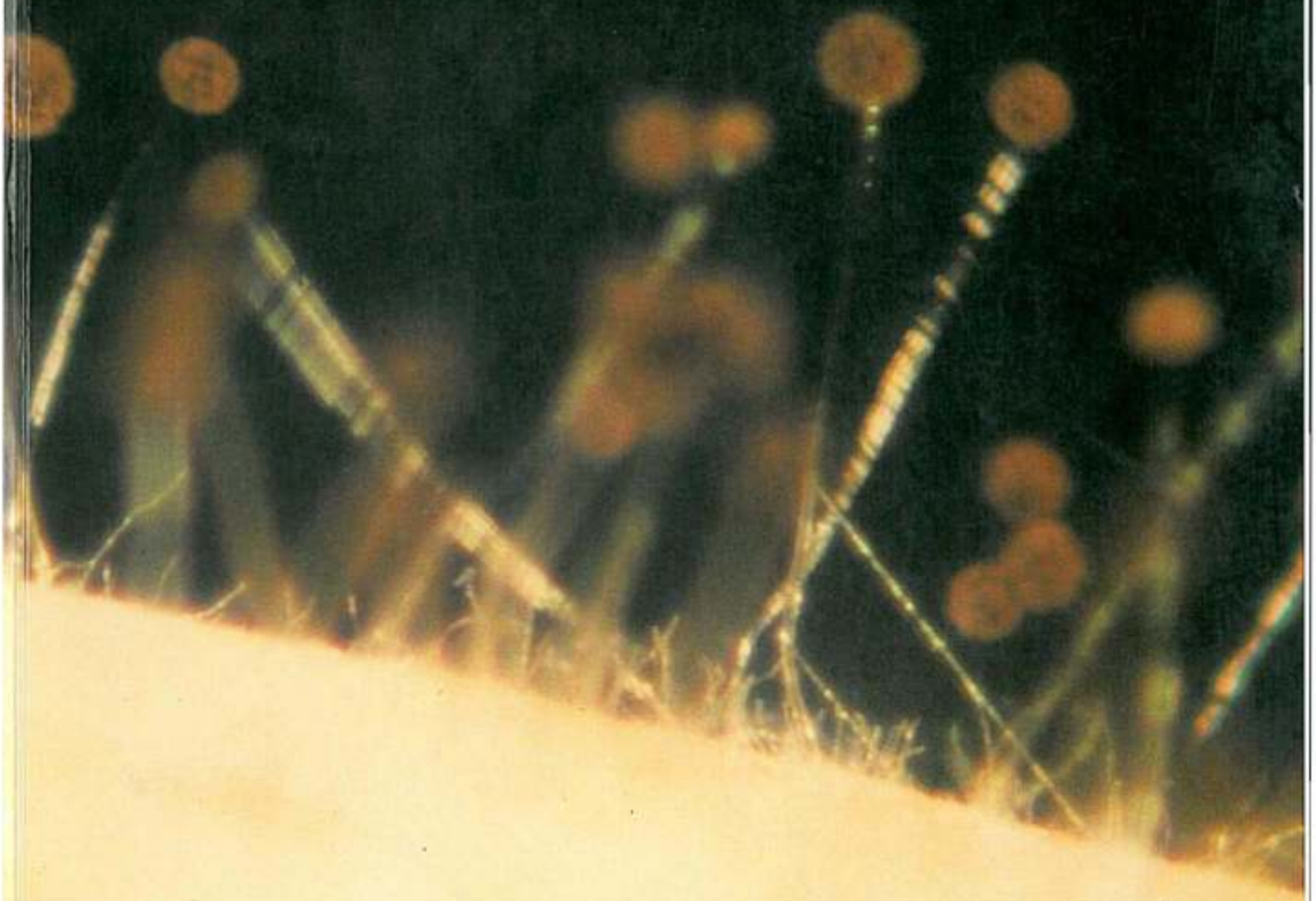
A Pictorial Guide to the Identification of Seedborne Fungi of Sorghum, Pearl Millet, Finger Millet, Chickpea, Pigeonpea, and Groundnut



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Abstract

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Seeds of sorghum, pearl millet, finger millet, chickpea, pigeonpea, and groundnut are known to harbor over 62 seedborne fungi belonging to 37 genera. In this bulletin, an attempt has been made to help agricultural scientists and students identify seedborne fungi, usually observed during the seed health tests conducted for phytosanitary certification of the six ICRISAT mandate crops This bulletin provides descriptions and illustrations of 45 seedborne
methods of seed transmission, detection, symptoms on the seed, morphological characteristics of the fungi, quarantine importance, and control measures to eradicate seedborne inoculum and to prevent inadvertent introductions. Microphotographs are included to help identify the fungi. A world list of seedborne diseases is also given to help regulatory agencies formulate policies involving seedborne fungi, so that unnecessary restrictions on the movement of disease-free germplasm can be avoided.

Resumé

Guide illustré d'identification des champignons transmis par les semences de sorgho, de mil, d'éleusine, de pois chiche, de pois d'Angole et d'arachide Les semences de sorgho, de mil, d'éleusine, de pois chiche, de pois d'Angole et d'arachide risquent de transmettre plus de 62 champignons appartenant à 37 genres. Ce guide vise à aider les chercheurs agricoles et les étudiants a identifier les champignons, transmis par les semences, généralement observés lors des tests effectués pour la certification phytosanitaire des six cultures du mandat de l'ICRISAT. On y trouvera les descriptions et les illustrations de 45 champignons transmis par les semences, ainsi qu'une information sur les maladies qu'ils causent, les methodes de transmission des semences, la détection, les symptômes sur les graines, les caractéristiques morphologiques des champignons, la quarantaine et les mesures à prendre pour détruire l'inoculum et prévenir son introduction par inadvertance. Des photomicrographies sont incluses pour faciliter l'identification des ces champignons. Une liste mondiale des maladies transmises par les semences est aussi présentée, afin d'aider les agences habilitées à formuler une réglementation sur les champignons transmis par les semences et ainsi éviter l'imposition de restriction non nécessaires sur le mouvement de ressources phytogénétiques exemptes de maladie.

Resumen

Una guía pictórica de los hongos albergados en las semillas de sorgo, mijo, garbanzo, guandul y maní. Se conoce que las semillas de sorgo, mijo, garbanzo, guandul y maní albergan mas de 62 hongos pertenecientes a 37 géneros. En este boletín, se ha intentado ayudar a los científicos y estudiantes de agricultura a poder identificar los hongos albergados en las semillas que se suelen observar durante las pruebas de las semillas hechas para la certificación fitosanitana de los seis cultivos mandatonos de ICRISAT. Este boletín facilita descripciones e ilustraciones de 45 hongos albergados en las semillas con una información breve sobre las enfermedades por ellos cuasadas, los métodos de transmision de las semillas, la detección, las síntomas de las semillas, las características morfológicas de los hongos, importancia de cuarentena, y medidas de control para erradicar los inóculos albergados en las semillas y evitar las introducciones inadvertidas, se incluyen microfotografias para facilitar la identificación de los hongos. Tambien se encuentra una lista mundial de las enfermedades albergadas en semillas a fin de que las agencias reguladores puedan formular políticas con respecto a hongos de las semillas para que las restricciones innecesanas sobre el movimiento de germoplasma libre de enfermedades se puedan evitar.

الخلاصة

موجه مصور لتعيين فطر ناتج عن البذور لمرغوم و دخن لؤلؤي و دخن اصبع و حمص وبسلة هندية وفول سوداني : من المعلوم ان بذور سرغوم و دخن لؤلؤي و دخن اصبع و حمص وبسلة هندية و فول سودانيالبذور المعقودة للتصديق على صحة ٦ غلال مستدبة للمعهد . ويقدم البلاغ وصف ٤٥ فطرا ناتجا عن البذور و تصويره مع اعلام موجز بشان العلات المسببة منه ، وكذلك طرق نقل البذور و كشفها ، كما يبين علاماتها و ميزات تشكلية لفطر واهمية محجر صحي / كرنتيتا و اجراءات الضبط لابادة لقاح فطر ناتج عن البذور و لمنع ادخالات غير متعمدة . ثم يضمن التصاوير الدقيقة لمساعدة تعيين الفطر . وكذلك يقدم قائمة الكلمات لعلات ناتجة عن البذور لمساعدة الوكالات التنظيمية في وضع سياسات متضمنة فطرا ناتجا عن البذور للتجنب من القيود غير الضرورية على حركة الجبلة الجرثومية الحرة من العلات .

Cover: Growth of *Aspergillus niger*, a pathogen of groundnut, after incubation, showing conidiophores with conidial heads in situ.

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1993

Acknowledgments

The assistance of Dr B.C. Sutton, Head of Taxonomic and Identification Services, and his colleagues at the International Mycological Institute, UK, in identifying the fungi; and Dr K.S. Varaprasad, Officer-in-Charge, Plant Quarantine Regional Station, National Bureau of Plant Genetic Resources, Hyderabad and Dr S.D. Panchbhai, Plant Pathologist, Mahyco, Jhalna, India, in providing support, is gratefully acknowledged.

Contents

Erratum

Pages 142-144 and 165.

For *Leptosphaerulina crassiaca* (Sechet.) C.R. Jackson & D.K. Bell.

please read *Leptosphaerulina crassiasca*

Foreword

The International Crops Research Institute for the Semi-Arid Tropics (ICRISAT) aims to improve the grain yields and quality of sorghum, pearl millet, finger millet, chickpea, pigeonpea, and groundnut in countries of the semi-arid tropics. ICRISAT also serves as a world repository for the genetic resources of these crops.

The Government of India has agreed to the unrestricted exchange of the germplasm of its mandate crops by ICRISAT, subject to quarantine regulations. For safe germplasm exchange, the Indian Government and ICRISAT require safeguards to be followed, not only by ICRISAT scientists at all the Institute's locations, but also by cooperators in various countries. Details of the certification necessary for safe exchange are available from the Plant Quarantine Unit at ICRISAT Center.

Plant quarantine work at ICRISAT involves the phytosanitary examination of the seeds and plant material of the Institute's six mandate crops prior to their export, and examination of any imported material. These examinations are carried out in close collaboration with the plant quarantine authorities of the Government of India. Since 1974 the Institute has exported 944 044 seed samples to 153 countries, and has imported 171 826 samples from 98 countries.

The unrestricted movement and exchange of germplasm is vital to progress in crop improvement programs, but such movement must not at any time jeopardize crops by spreading pests or diseases. High priority should be given to designing treatments or other procedures necessary to allow movement of seeds without spreading diseases. The international exchange of disease-free germplasm of ICRISAT's six crops should be possible if safeguards are applied at ports of entry and exit. Quarantine stations receive seeds for certification from various sources including seed industries, growers, research stations, individual scientists, traders, etc. The risk of spreading seedborne pathogens varies widely with seed source. When the seed is received from scientists, research stations, and well-equipped seed industries, these risks are greatly reduced because it is assumed that the crop has been monitored during its active growing season by a qualified pathologist.

The authors, during the course of their quarantine work at ICRISAT, have intercepted 45 seedborne fungi out of the 62 reported in the literature; these 45 fungi are illustrated in this publication. It is hoped that this publication will provide useful information to agricultural scientists and regulatory agencies dealing with the import and export of seed and plant material, and will help to minimize the risk of spreading diseases that could reduce crop yields and limit the amount of food farmers can produce.

The authors would welcome communications on any inadvertent omissions, or additional data.

Y.L. Nene
Deputy Director General, ICRISAT

Introduction

Coverage

Fungi that infect seeds and survive as spores or resting structures on and within seeds are known as seedborne. The term as used in this publication refers to the association of pathogenic fungi which are either transmitted by, or transported with seed. Seed-transmitted fungi often produce infected plants or can be the focus of inadvertent introductions; whereas seed-transported fungi are those which are present on the seed, but have not been shown to cause subsequent infection. Seed-transported fungi are considered relatively unimportant in crop production, but do represent one way by which a pathogen can be introduced into an area from which it was originally absent. Both types of fungi are important for plant quarantine and to plant pathologists, and no distinction is made between seed-transmitted and seed-transported fungi in this publication.

By applying the above criteria to pathogens that affect the the mandate crops of ICRISAT, 62 fungi belonging to 37 different genera were found to have been reported as seedborne, and to cause various diseases under field conditions; these fungi are listed in Appendix 1. Seedborne fungi, which predominantly occur as pathogens causing grain molds, have been excluded, because such fungi infect seed during storage, transportation, etc., through their omnipresent spores. The seedborne inoculum of such fungi is less important than the inoculum from infected seedlings or plants.

International Seed Health Testing Methods (ISHTM) are used to detect the presence of seedborne fungi on seeds. It is recommended that when testing seeds for quarantine and phytosanitary certification, these standardized procedures should be adopted, as this will help to eliminate situations where examinations in a receiving country reveal discrepancies with health certificates accompanying the seeds. Three ISHTMs are briefly discussed in Appendix 2.

The characteristic morphological features of 45 seedborne fungi belonging to 26 genera that affect the six ICRISAT mandate crops, and that can be seen on infected seed when tested using ISHTMs are described in this publication. Among these, 11 species belonging to 10 genera occur on more than one ICRISAT crop; their descriptions and illustrations are included for the crop on which the fungus is most frequently recorded. However, to show pathogen variability, illustrations of infected seeds of the other susceptible ICRISAT crops have also been included. This does not imply that isolates of one crop can infect the other, or vice versa.

This publication can only provide a glimpse of the total range of morphological diversity exhibited by seedborne fungi, it aims to help the user quickly identify a fungus by comparing the symptoms shown in the photomicrographs with those exhibited by the seed under examination. Differences in colony characters, conidial morphology, and ornamentation of fruiting bodies which can be seen under the stereoscopic microscope are used to differentiate between the various genera. Fungal characters visible under a compound microscope are used to identify

species. Whenever possible, it is advisable to confirm the identity of the fungus with the help of professional mycologists, and to prove its pathogenicity.

Control

Control measures that should be followed before crop seeds are exported are given at the end of each description. These control measure only relate to seed health management. Some of the fungicides mentioned by their trade names in the text may not be available in some countries; in such case alternative fungicides should be used, and to help select the most appropriate their active ingredient has been mentioned. In Appendix 3 a standardized seed treatment schedule followed at ICRISAT Center is given. This treatment schedule is updated as and when more information on the biology and control of a fungus becomes available.

Photographs

The photographs depict typical macro- and microscopic fungal structures, and were taken using stereobinocular and compound microscopes. Naturally infected seeds were observed under a Wild M7S stereobinocular microscope with a ×6 to ×31 zoom magnifier, fitted with an interchangeable ×2 additional objective, and ×10 and ×15 eye pieces. The microscope was fitted on a bright/dark, field-transmitted light stand with a 6v/10w halogen bulb. Two additional 6v/20w incident light illuminators were used to obtain shadow-free illumination. The camera used was a Wild with a 35 mm back ×0.32 photomagazine connected to a Wild photoautomat MPS 45. Kodak ET-135-36 tungsten color film, 160 ASA was used. The pictures were taken by adjusting the photoautomat to 400 ASA.

To prepare material for compound microscopy, seeds were inoculated with type cultures and incubated on moist blotting paper in plastic petri dishes under near ultra violet (NUV) light for a 12-h light/dark regime for 7 days at 22 ± 2°C. Squash mounts of fungal growth on seed surfaces were made in sterile water without staining, and observed under a compound microscope. The microscope used was an Olympus BH2 fitted with ×20, ×40, and ×100 apochromat objectives, and a ×10 eye piece. The microscope was fitted with an Olympus PM10 AD camera system. Kodak 35 mm black and white film, 32 ASA was used. Rates of magnification are provided for each photograph in its caption.

Sorghum

Acremonium strictum W. Gams

Acremonium strictum causes sorghum wilt. Infected seed can be identified by visual examination and incubation tests. Infected dry seeds are shrunken, desiccated, mummified, and misshapen (Fig.1A). Such seeds weigh less than healthy seeds, and germinate poorly. The fungus grows on incubated seed as a white mycelium that produces conidia (Figs.1B and 1Ca). When the seed is incubated for 8–21 days, the perithecial state of the fungus, which belongs to the genus *Neocosmospora*, is formed on the seed (Fig.1Cb). Colonies in the conidial state appear similar in color and texture to some species of *Fusarium*. The **mycelium** consists of thin, delicate, hyaline, smooth, branched hyphae. Hyphal cells are usually 0.5–1.5 μm wide. Often several hyphae aggregate to form thick hyphal strands (Fig.1Da) on which small conidiophores bearing conidia are formed. **Conidiophores** (Fig.1Db) are simple, whorled or cymoid, hyaline and subulate, 32.5–60 × 1–1.8 μm, and arise directly at right angles from the aggregated thick strands of vegetative hyphae in a basitonous fashion. **Conidia** (Fig.1Dc) are hyaline, single-celled, and produced endogenously at the apex of the conidiophore in false heads. They are ellipsoidal to cylindrical with rounded ends, straight or slightly curved, and measure 2–5 × 1–1.5 μm. **Perithecia** are usually orange to red, rarely dark brown, globose to pyriform, ostiolate with a short neck lined with periphyses, and contain several asci. These **asci** (Fig.1Dd) are hyaline, cylindrical, rarely clavate, with a short stalk, each containing eight ascospores in a uniseriate fashion, and are 80–104 μm long. **Ascospores** (Fig.1De) are pale brown to almost hyaline, single-celled, globose to ellipsoidal, thick-walled with irregular, rough surfaces, and are 13–20 μm in diameter (Gams 1968; Frederiksen 1984; Cannon and Hawksworth 1984; Bandyopadhayay et al. 1987).

Control. Control measures to be applied during phytosanitary inspections should aim to exclude the seedborne inoculum, by collecting seeds from disease-free plants. Information on seed treatment to eradicate the seedborne inoculum of the fungus is not available.

Figure 1A.
Dry seeds of sorghum showing damage caused by Acremonium strictum *(right), and healthy seeds (left), ×11.*

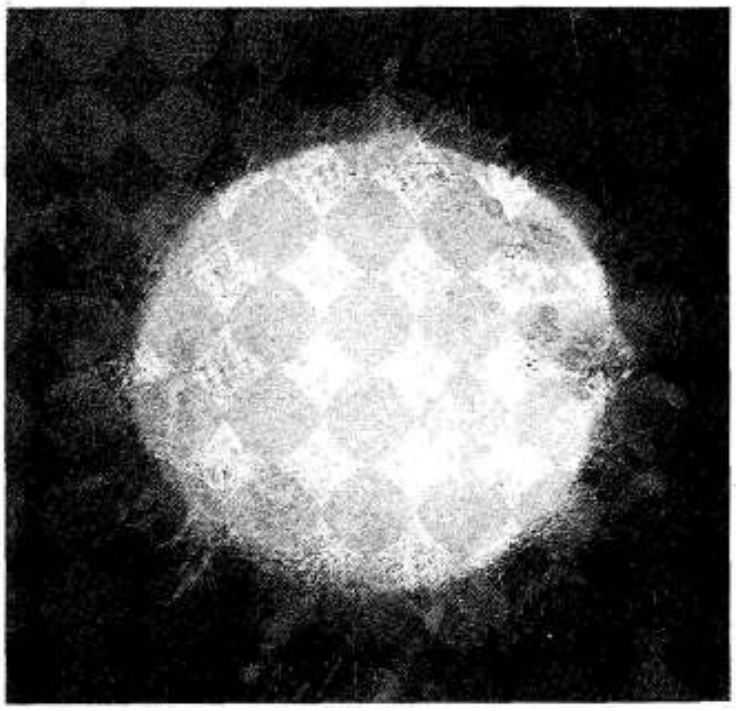


Figure 1B.
Infected seed after incubation, showing the growth of A. strictum, *×22.*

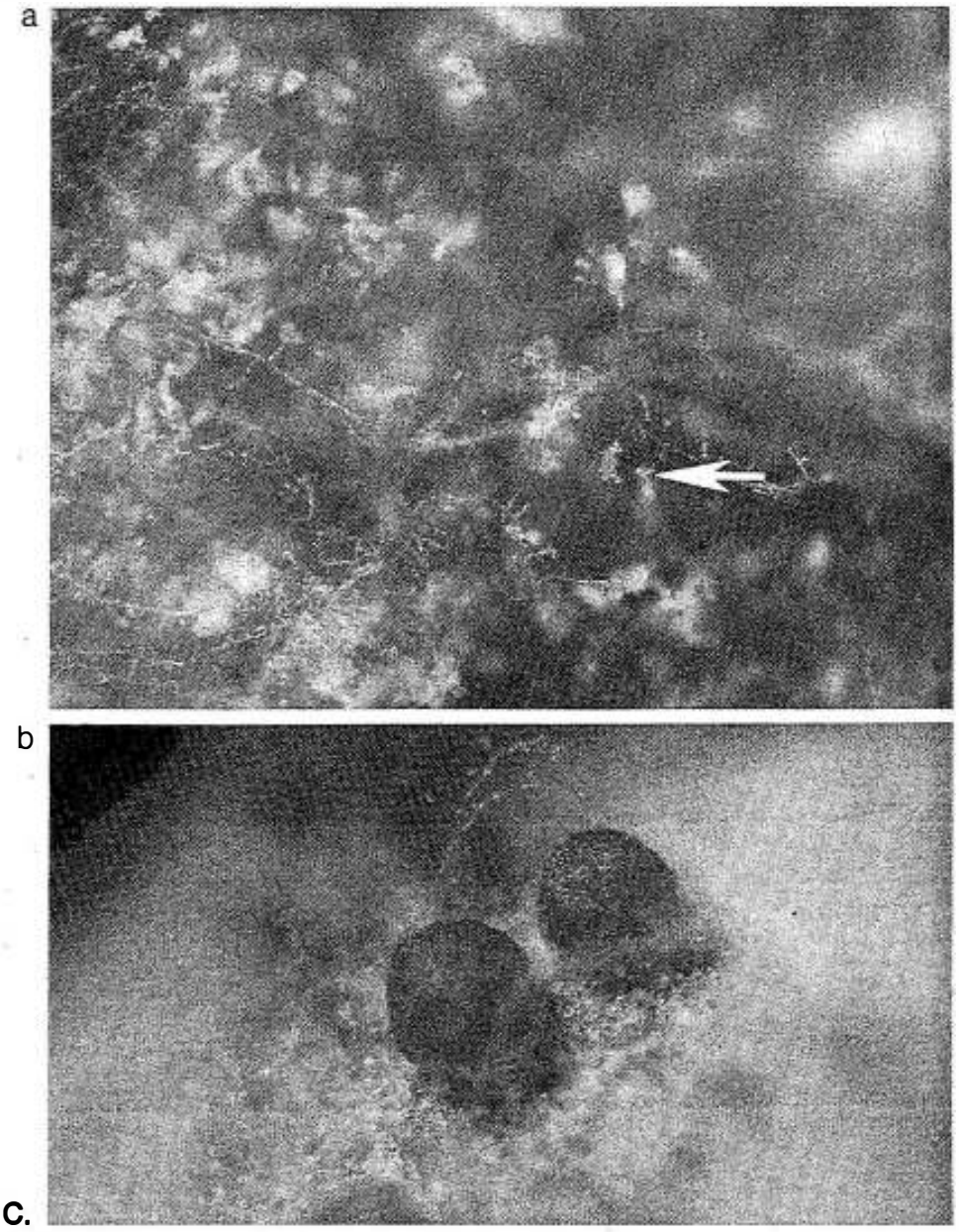
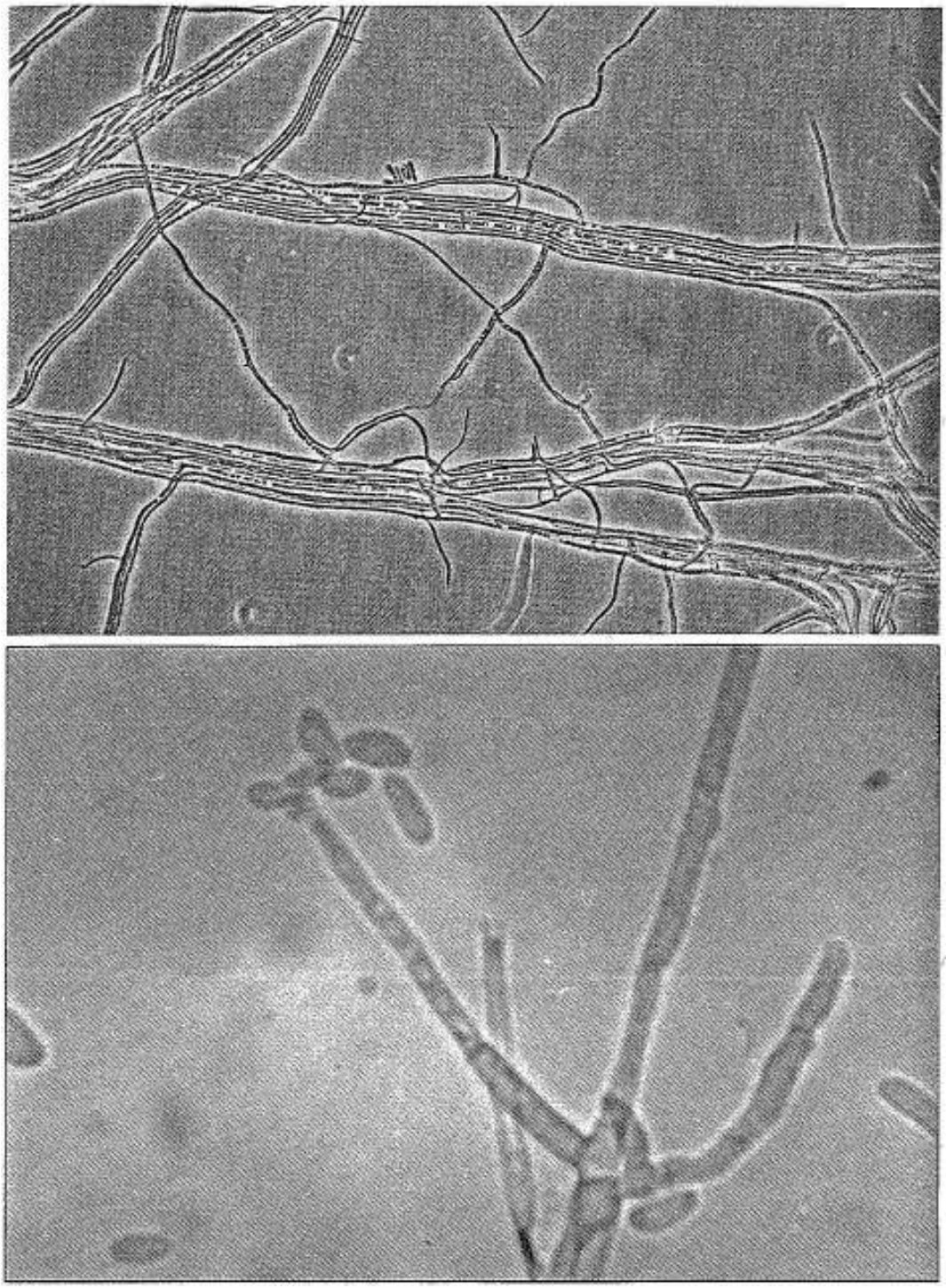


Figure 1C.
(a) Conidial heads (arrowed) of A. strictum, *×75,*
(b) perithecia produced by Neocosmospora *sp, ×75.*



a

b

Figure 1D
(a) Aggregated hyphal strands, ×451, (b) conidiophores, ×1130,

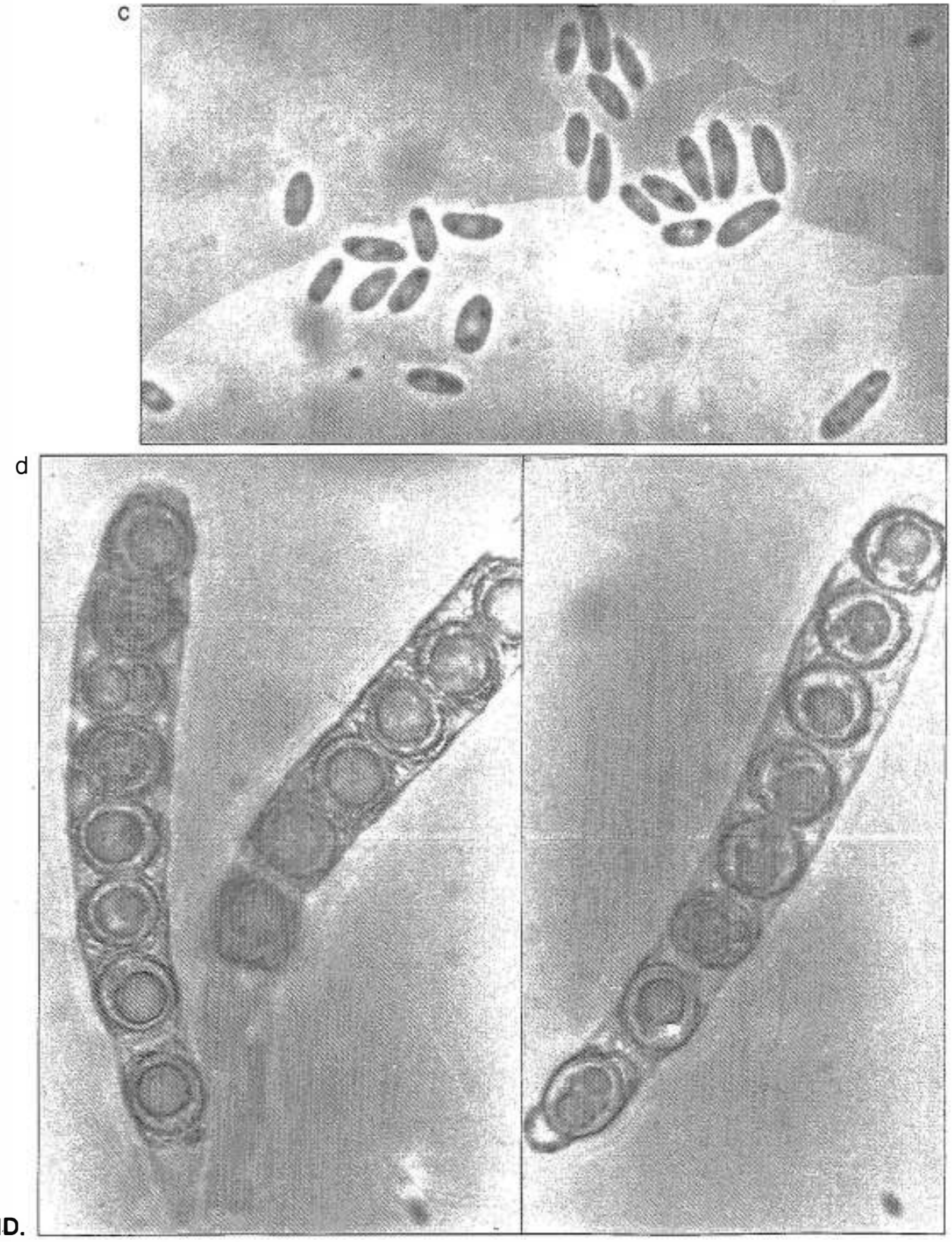


Figure 1D.
*(c) conidia (*A. strictum*), ×1130, (d) asci, ×1130,*

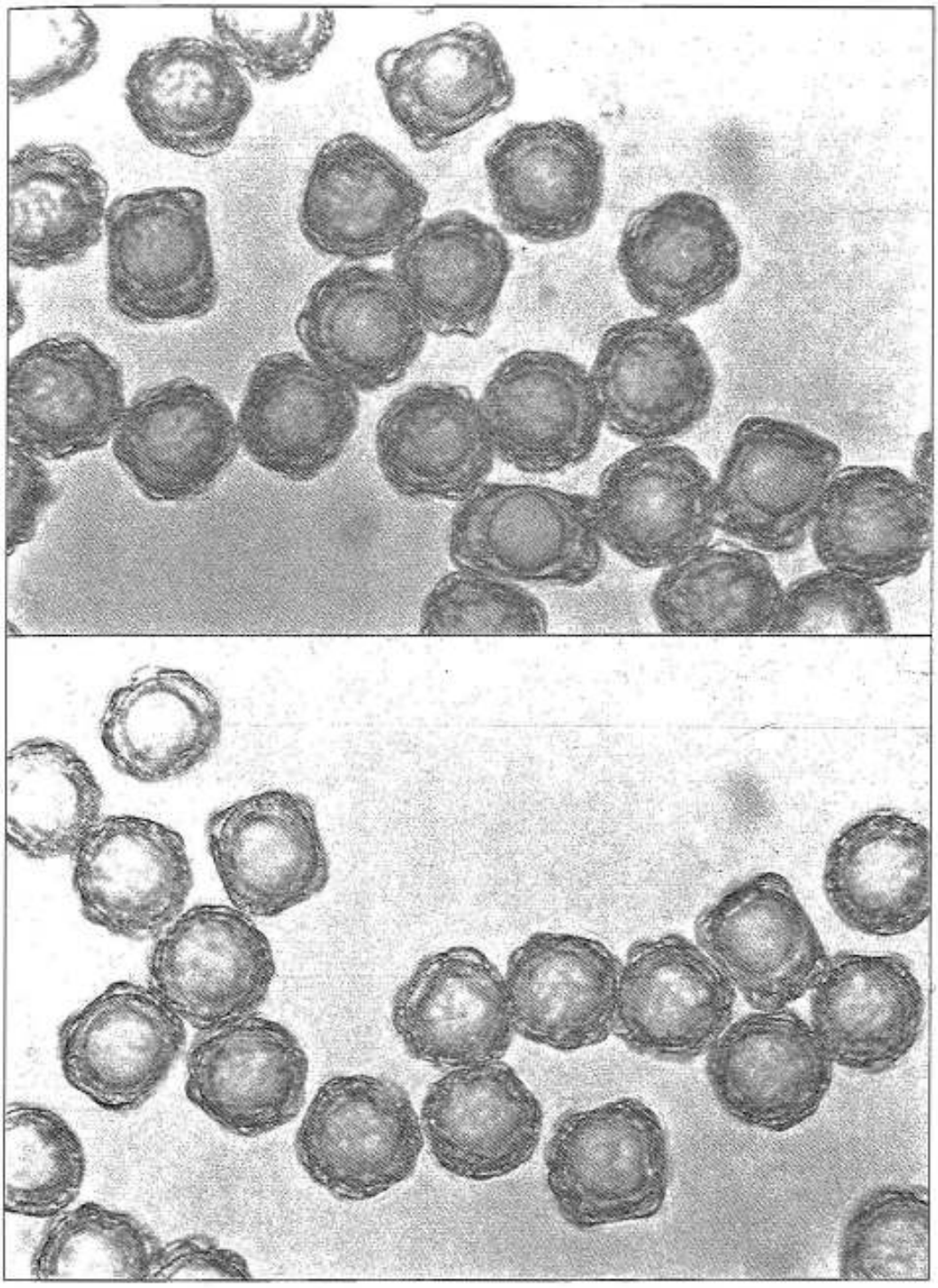


Figure 1D.
*(e) ascospores (*Neocosmospora *sp), ×1130.*

Bipolaris sorghicola (Lefebvre & Sherwin) Alcorn

Helminthosporium sorghicola Lefebvre & Sherwin

Drechslera sorghicola (Lefebvre & Sherwin) M.J. Richardson & E.M. Fraser

Bipolaris sorghicola causes sorghum target leaf spot. Infected seeds can be detected by incubation tests (Fig.2A). The fungus grows on the incubated seed producing mycelium, condiophores, and conidia. **Conidiophores** usually arise singly or in small groups on the infected seed. They are straight, or flexuous with a swollen base and a geniculated apex, smooth, simple, septate, 115–700 µm long, and 6–9 µm wide. They usually bear conidia in an acropleurogenous fashion (Fig. 2Ba). The most distinguishing character of this species is that the primary conidia, while still attached to the conidiophore, frequently bear long secondary conidiophores on which small secondary conidia are produced (Fig.2Bb). **Conidia** are 3–8 in number, distoseptate, 30–100 × 12–19 µm, pale to mid-golden brown, slightly curved, fusiform, ellipsoid or straight, with an inconspicuous hilum (Fig.2C). Conidia are frequently seen attached to the conidiophores (Tarr 1962; Ellis and Holliday 1976; Sivanesan 1987).

Control. Seed treatment with ferbam at about 2.5 g kg^{-1} is effective in reducing seedborne inoculum (Neergaard 1979).

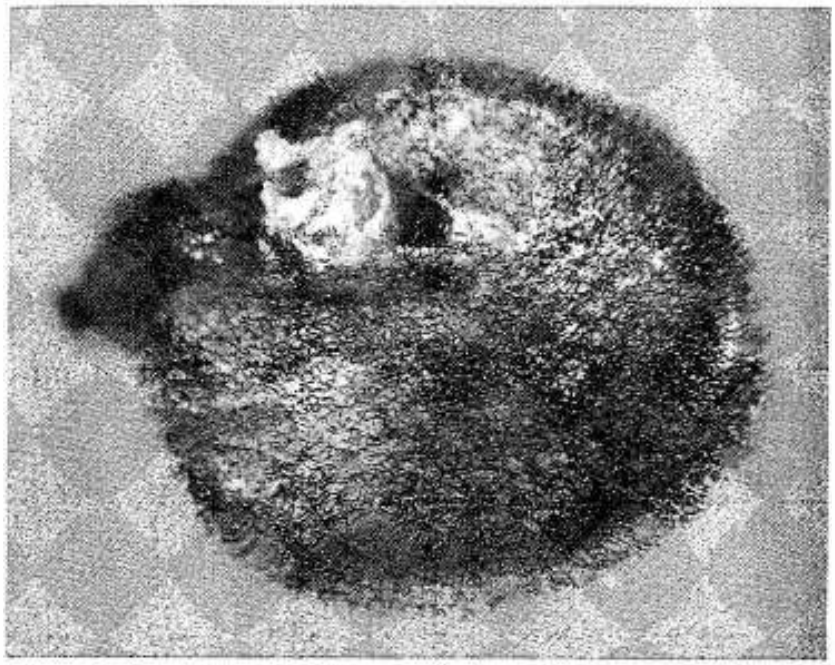


Figure 2A.
Infected sorghum seed after incubation, showing the growth of Bipolaris sorghicola, *×27.*

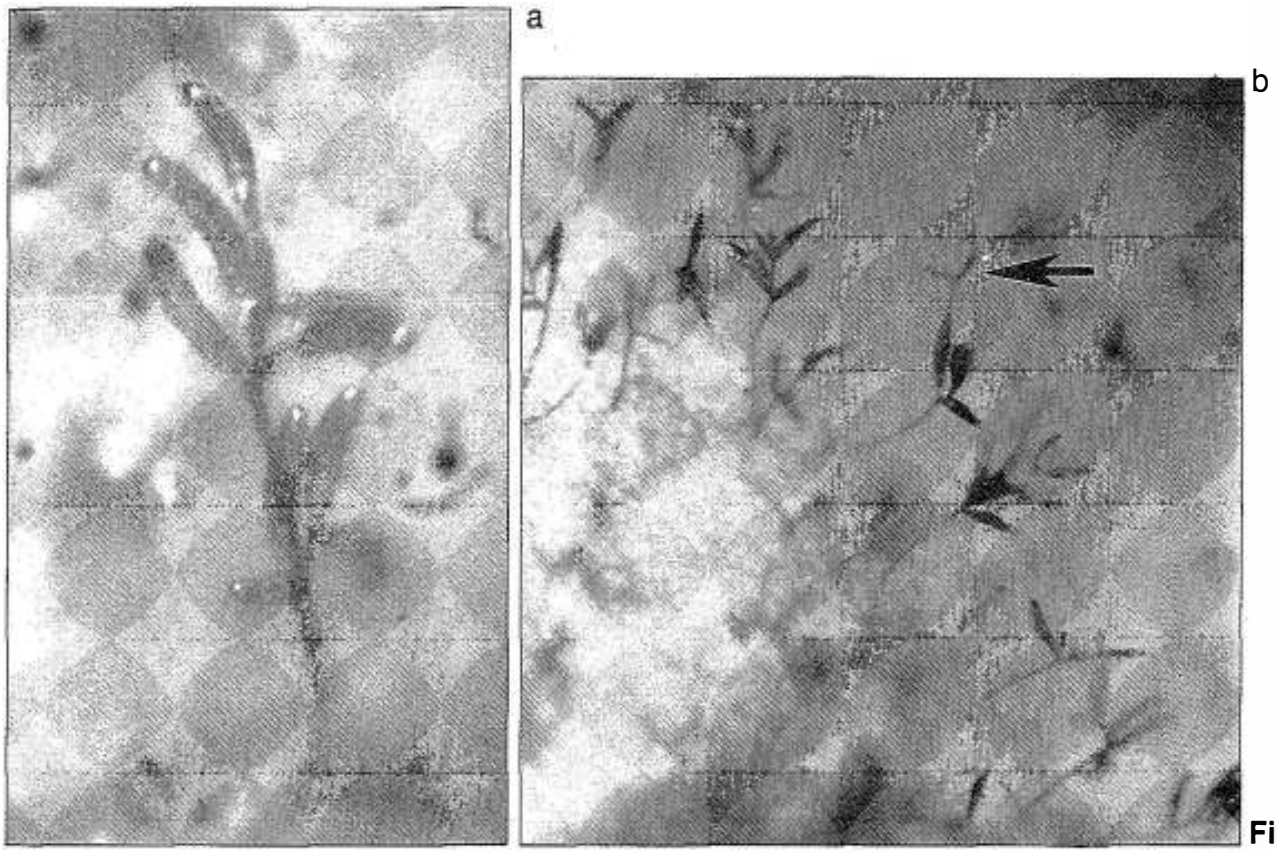


Figure 2B.
(a) Conidiophore and conidia, ×122, and (b) secondary conidia (arrowed) produced on the secondary conidiophores by B. sorghicola, *×113.*

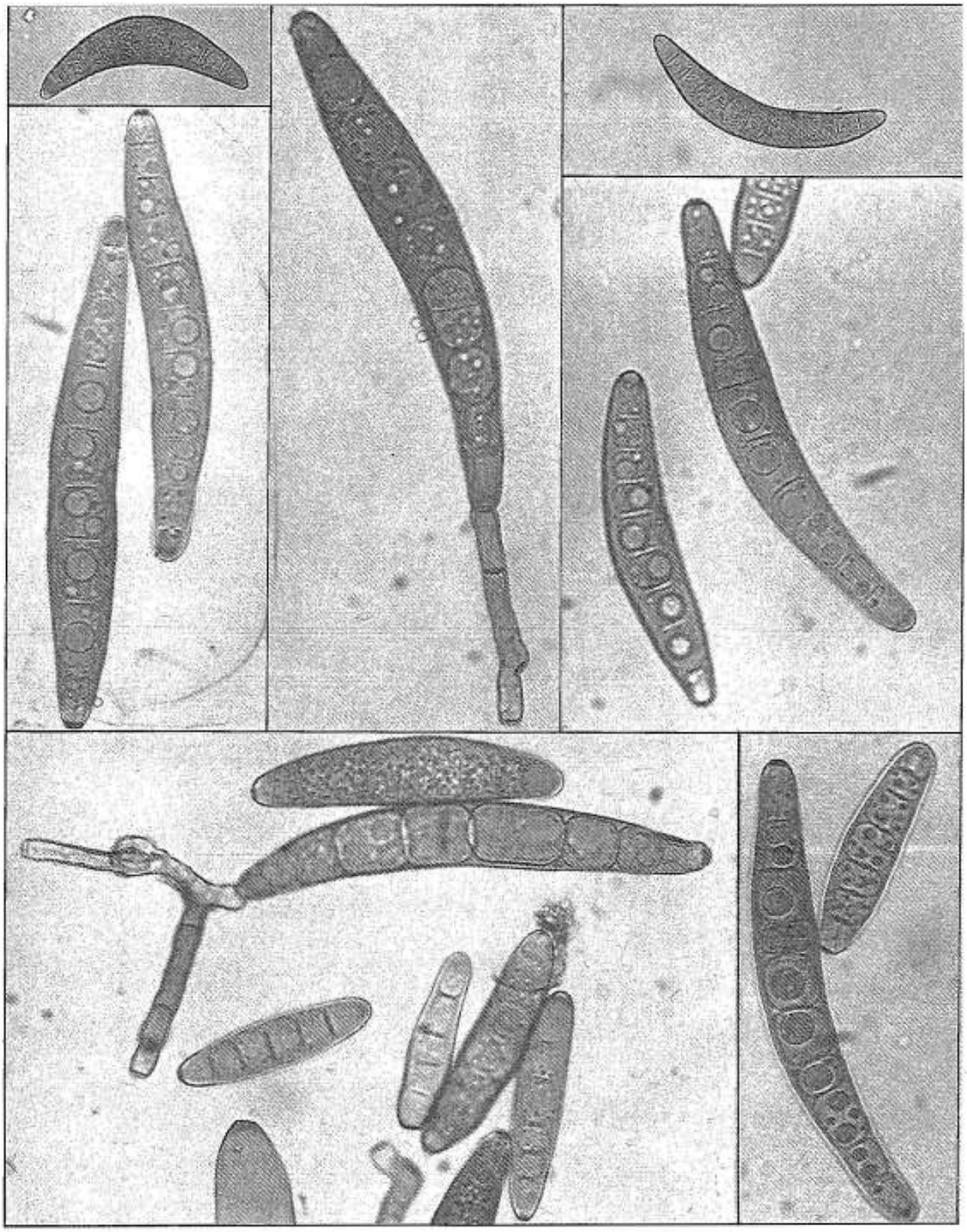


Figure 2C.
Conidia produced by B. sorghicola, *×677*.

Cercospora sorghi Ellis & Everhart

Cercospora sorghi causes gray leaf spot of sorghum. The fungus is present as a dormant mycelium on and within infected seed, and can be detected by incubation tests. The fungus produces numerous light to dark gray conidiophores on the incubated seed. These conidiophores bear hyaline conidia which when massed together give infected seed a light grayish, velvety appearance (Fig. 3Aa). **Conidiophores** emerge from a stroma-like body on the surface of the seed, in small scattered tufts of three to four, or more (Fig. 3Ab). They are medium to dark brown, rarely pale, narrower towards the apical tip, irregular in width, simple but long, flexuous, and geniculate with each conidiophore bearing one to six conidia. Conidiophores are 20–80 × 3–5.5 µm, or even up to 150 µm long (Fig. 3B). The **conidia** are indistinctly multiseptate (1–12 septa), hyaline, 30–300 × 2–4 µm, acicular, obclavate to cylindrical, straight or slightly curved, with a truncate or obconically truncate base (Fig. 3B) (Tarr 1962; Mulder and Holliday 1974).

Control. Conduct pre-export crop health inspections during crop growth and select seeds from healthy noninfected fields. Fungicides such as benomyl, thiram, or chloranil can be applied at about 2.5 g kg^{-1} as seed dressings to eradicate seedborne infection (Neergaard 1979).

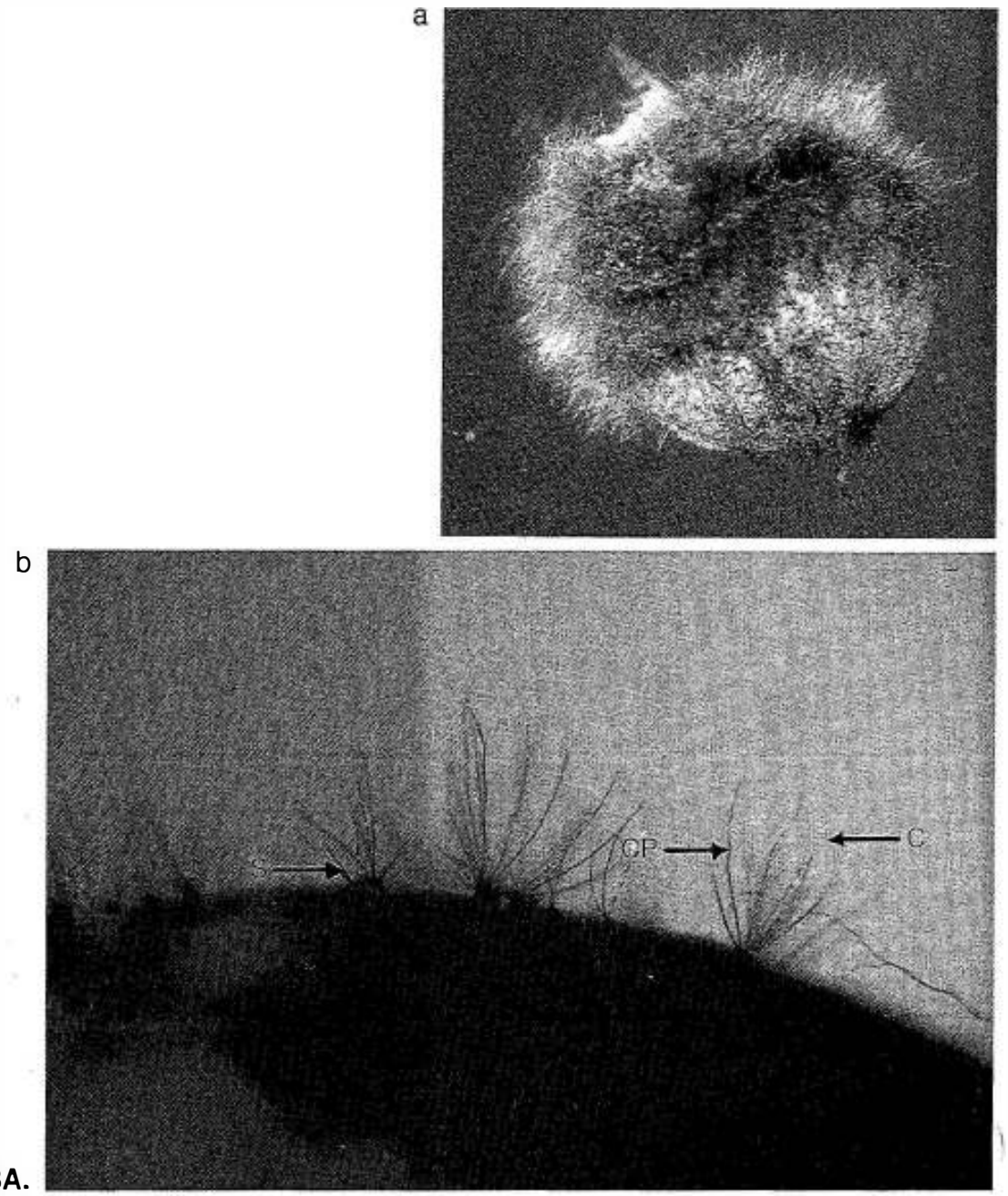


Figure 3A.
(a) Infected sorghum seed after incubation, showing the growth of Cercospora sorghi, ×*12, (b) stroma (S), conidiophores (CP), and conidia (C) of* C. sorghi, ×*75.*

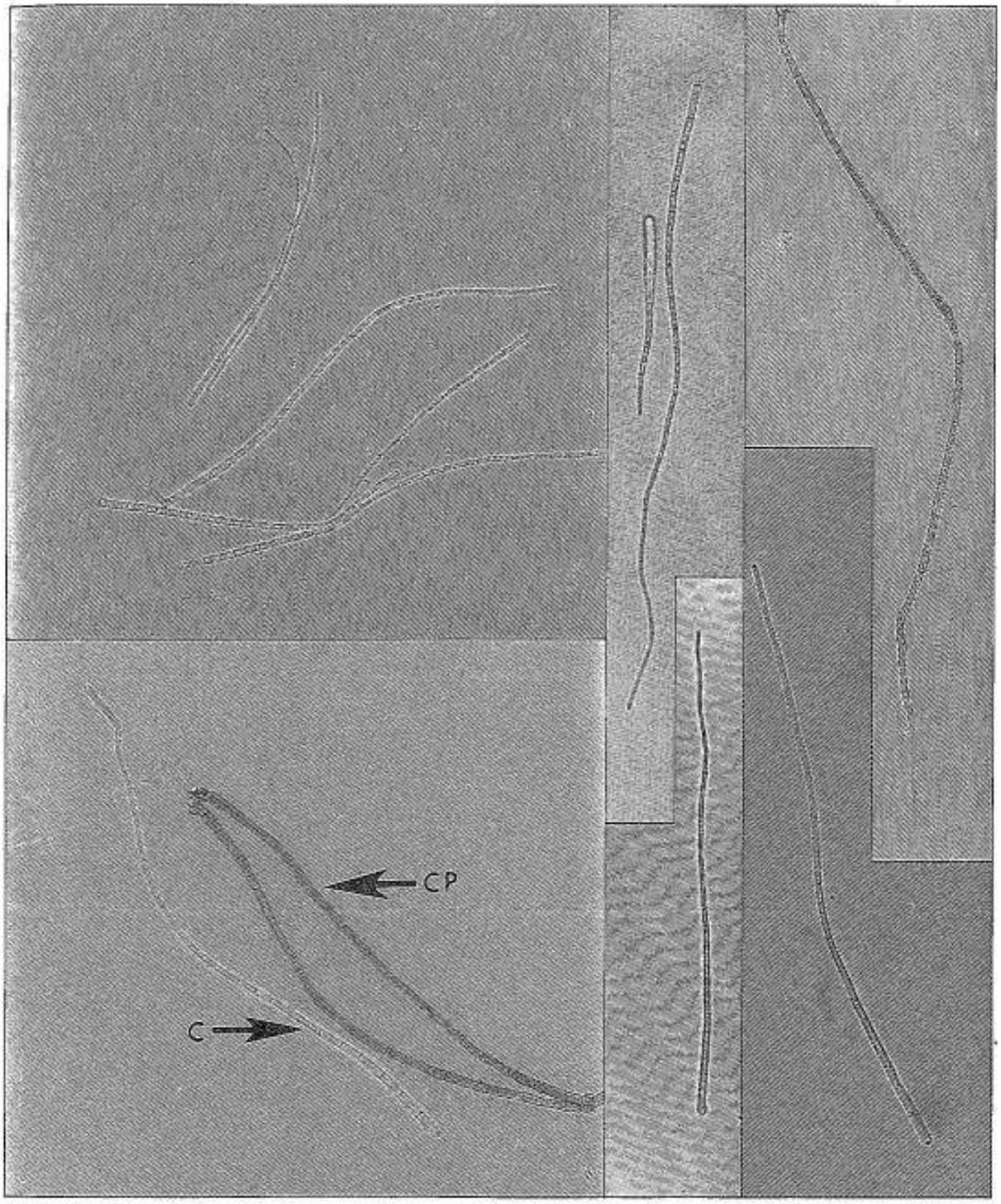


Figure 3B.
Conidiophores (CP) and conidia (C) of C. sorghi, *×448.*

Claviceps sorghi Kulkarni et al.

Claviceps sorghi causes ergot of sorghum. The fungus converts individual florets in infected panicles into hard sclerotia of varying sizes and shapes. These sclerotia are overwintering bodies that can be detected during visual seed examination. If crops are sown with contaminated seed, the fungus is disseminated to new areas. The **sclerotia** are hard, cylindrical, elongated, slightly curved and horn like, cream-grayish or dark brown or black, 10–25 mm long, and 4–6 mm wide (Fig. 4) (Butler 1918; Tarr 1962; Doggett 1988).

Control. Do not export seed from endemic fields. Separate the sclerotia manually by immersing seed lots in 10% common salt solution (Wallace and Wallace 1953). Seeds that do not float should be dried and then treated with the fungicides listed in Appendix 3.

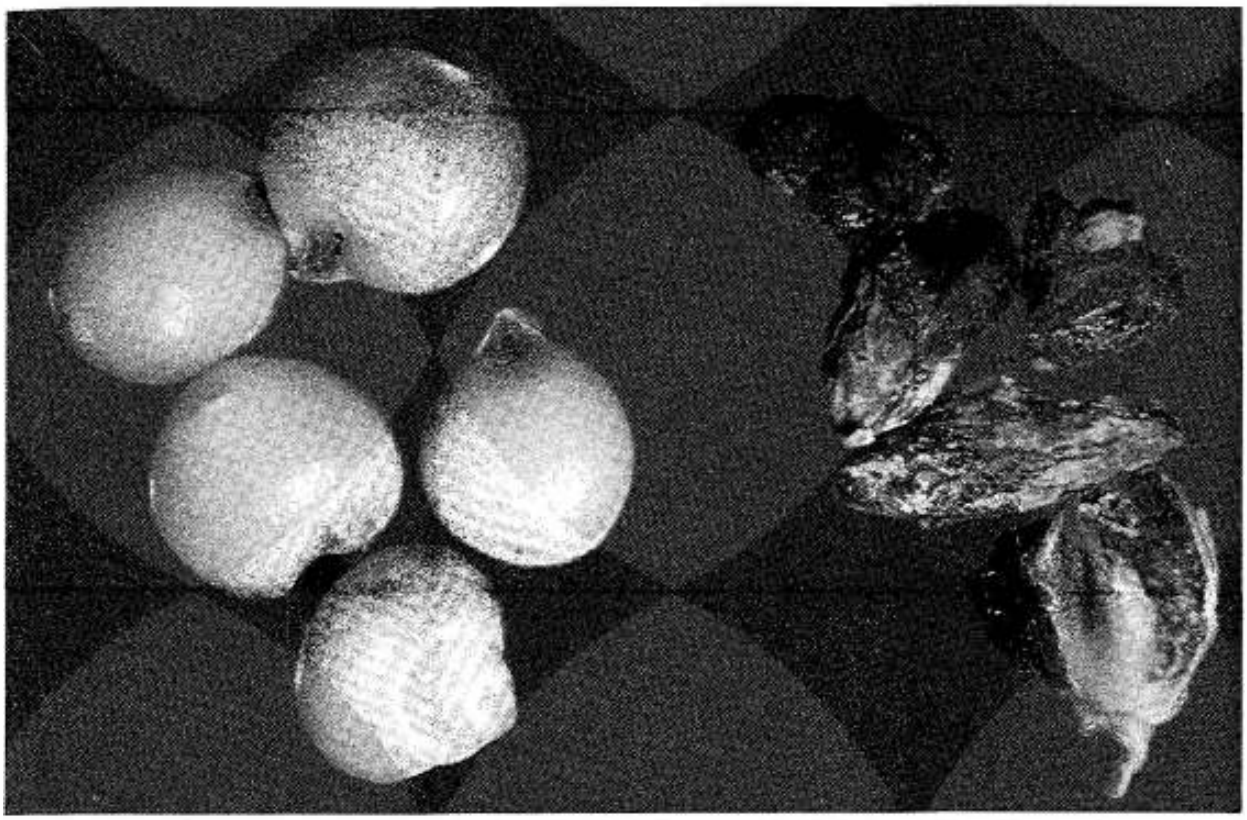


Figure 4.
Sclerotia of Claviceps sorghi *(right), and healthy sorghum seed (left),* ×*15.*

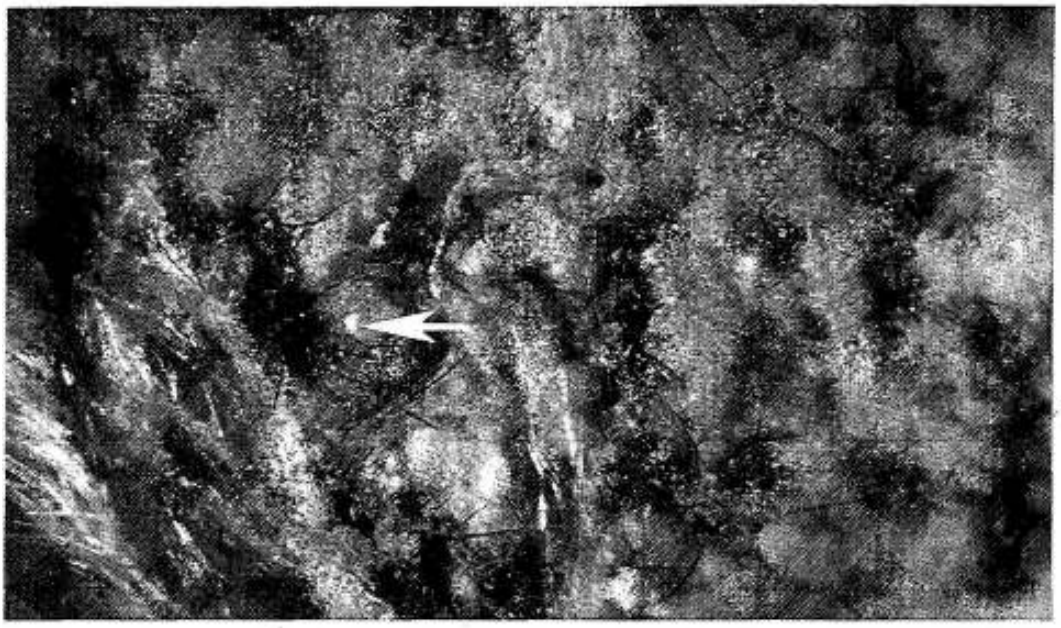
Colletotrichum graminicola (Cesati) G.W. Wilson

C. sublineolum Henn.

Anthracnose of sorghum is caused by *Colletotrichum graminicola*. The fungus can overwinter on seed as dry acervuli and within the seed as a dormant mycelium. Seeds harvested from diseased plants are likely to carry the fungus. Infected seeds can be detected by visual examination and incubation tests. Dry seeds show visible symptoms of infection, in the form of dark brown to black **acervuli** scattered on their surface (Fig. 5Aa). These acervuli are irregular in shape and consist of dark setae (Fig. 5Ab). Sometimes acervuli are also formed on the glumes. On incubated seed, the fungus produces numerous acervuli (Fig. 5B), which are rounded or elongate, separate or confluent, superficial, erumpent, with conspicuous multicellular, darkly pigmented setae, and 70–300 μm in diameter. The acervuli consist of a gelatinous or mucoid, salmon orange colored conidial mass (Fig. 5C). **Conidiophores** are hyaline, 8–20 μm long, and 4–8 μm broad (Fig. 5Da). Individual **conidia** are hyaline, single-celled, falcate, fusiform, spindle-shaped, with acute apices, and measure 19–28.9 × 3.3–4.8 μm (Fig. 5Db). **Setae** are brown with a dark swollen base and a pale rounded tip (Tarr 1962; Mordue 1967; Sutton 1980).

Control. Conduct pre-export crop health inspections during crop growth. Discard moldy seeds. Seed treatment with benomyl (Benlate® at about 2 g kg^{-1}) is advised to reduce the seedborne inoculum (Mishra and Siradhana 1978).

a



b

Figure 5A.
(a) Dry seeds of sorghum showing the symptoms of infection caused by Colletotrichum graminicola, *×6, and (b) dry acervuli (arrowed), ×75.*

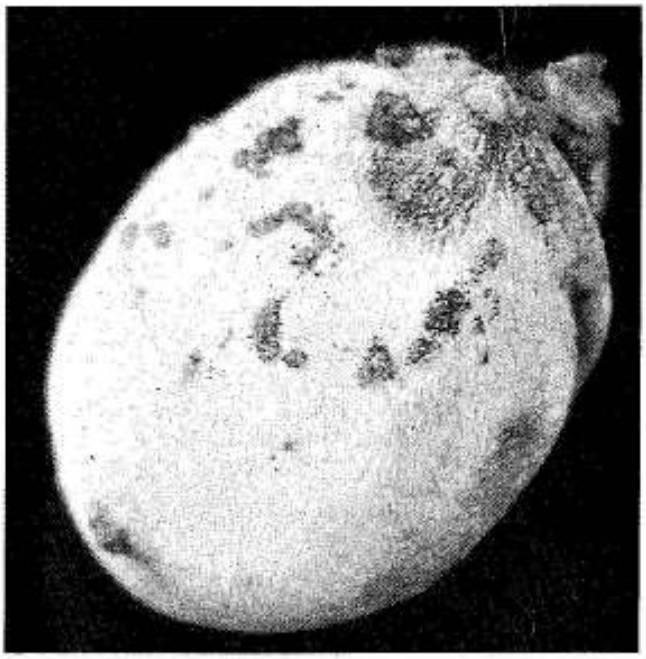


Figure 5B.
Infected sorghum seed after incubation, showing the growth of C. graminicola, *×32.*

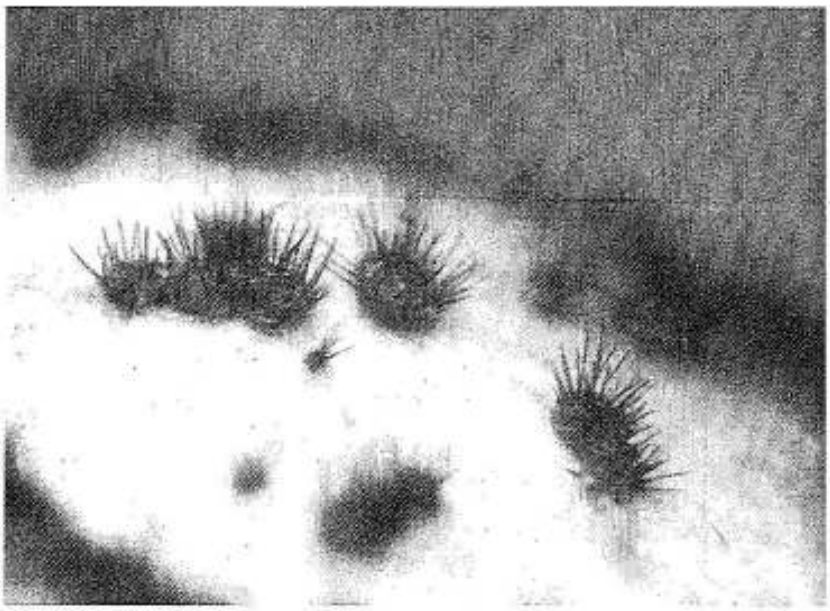


Figure 5C.
Acervuli containing conidial mass and setae of C. graminicola, *×75.*

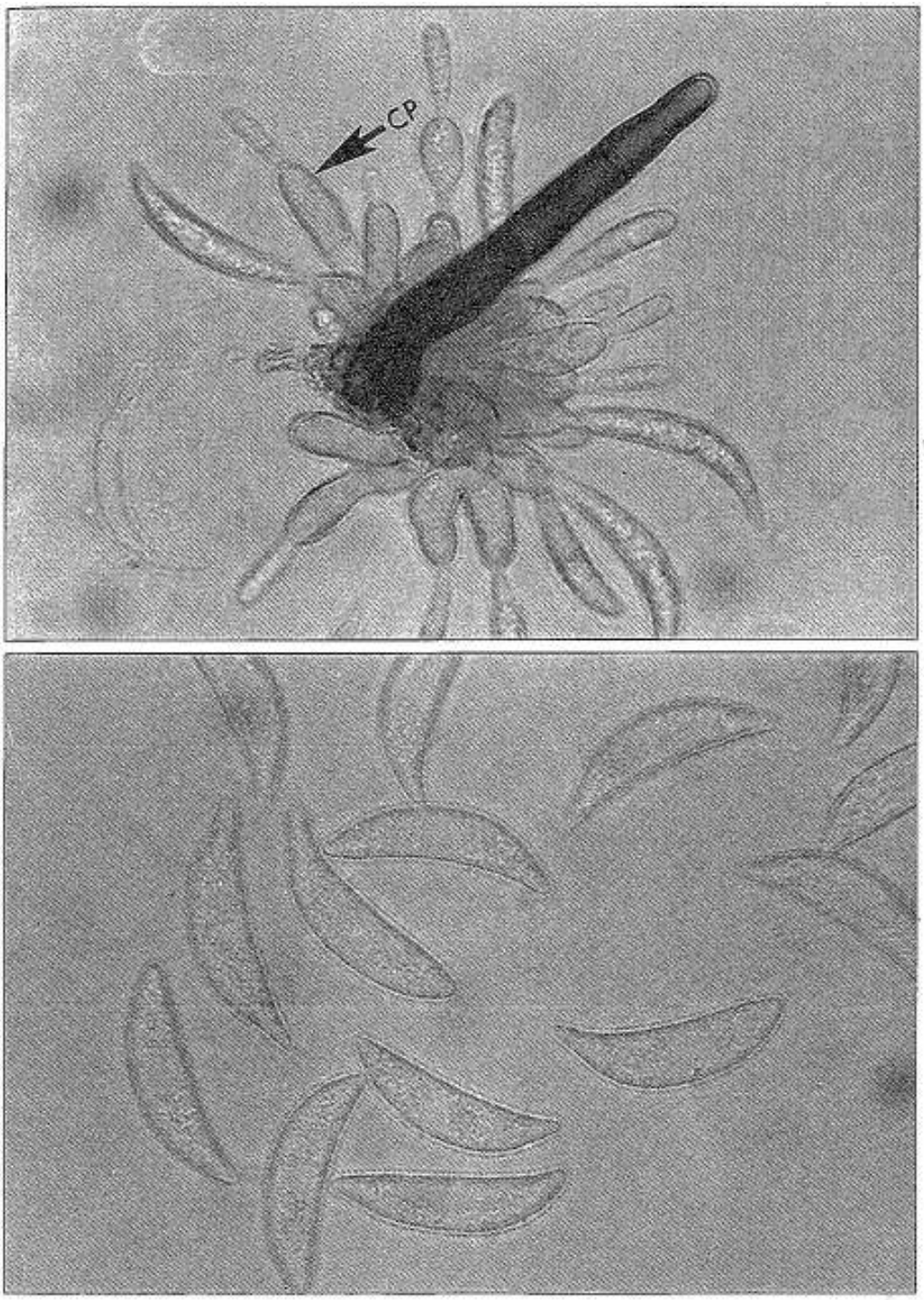


Figure 5D.
(a) Conidiophores (CP), ×1130, and (b) conidia of C. graminicola, *×1130.*

Exserohilum turcicum (Pass.) K.J. Leonard & E.G. Suggs

Helminthosporium turcicum Pass.

Bipolaris turcica (Pass.) Shoemaker

Drechslera turcica (Pass.) Subramanian & P.C. Jain

Exserohilum turcicum causes leaf blight of sorghum, and also attacks maize. Fungal isolates from sorghum and maize can attack either host singly or in combination. Seedborne infection can be detected by incubation tests. The fungus produces conidiophores and conidia throughout the seed surface (Fig.6A). The growth is hairy and is dark gray to brown. **Conidiophores** emerge in groups of two to six from the seed coat, and are 135–300 µm long, and 8–10 µm wide. They may be flexuous, simple, erect, with a swollen base and geniculated apex and bear typically straight and spindle-shaped conidia acropleurogenously (Figs.6B and 6Ca). Sometimes conidia are produced in loose spikes. **Conidia** are seen growing at the tips of the conidiophores, each of which continues to grow at the apex, pushing aside the first-formed conidium. Conidia are 3–11 distoseptate, measure 50–144 × 18–33 µm, pale to olivaceous brown, widest in the middle, fusoid to slightly curved, and taper towards both ends (Fig.6Cb). Conidia have a truncate and protuberant hilum in their basal cell which is visible as a small, thin stalk at the point of attachment to the conidiophore. This can be noticed at high magnifications even under the stereobinocular microscope (Ellis and Holliday 1971a; Chidambaram et al. 1973; Sivanesan 1987).

Control. Avoid collecting seed from endemic fields. Seed treatment during seed distribution, to minimize the risk of spreading the disease to hitherto noninfected areas, is important. Fungicides such as guazatine (Panoctine-R® at about 2 mL kg^{-1} of seed) are useful in eradicating superficial infection (Shree 1983). Captafol (Difolaton® at about 4 g kg^{-1}) eradicates both external and internal seedborne inoculum (Vidyasekaran et al. 1980).

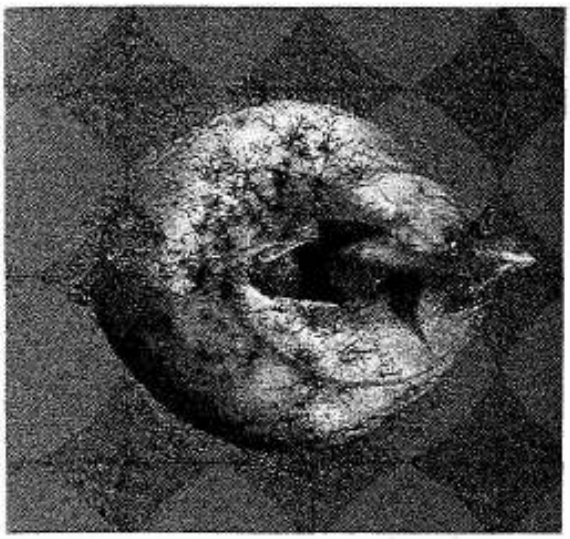


Figure 6A.
Infected sorghum seed after incubation, showing the growth of Exserohilum turcicum, *×18.*

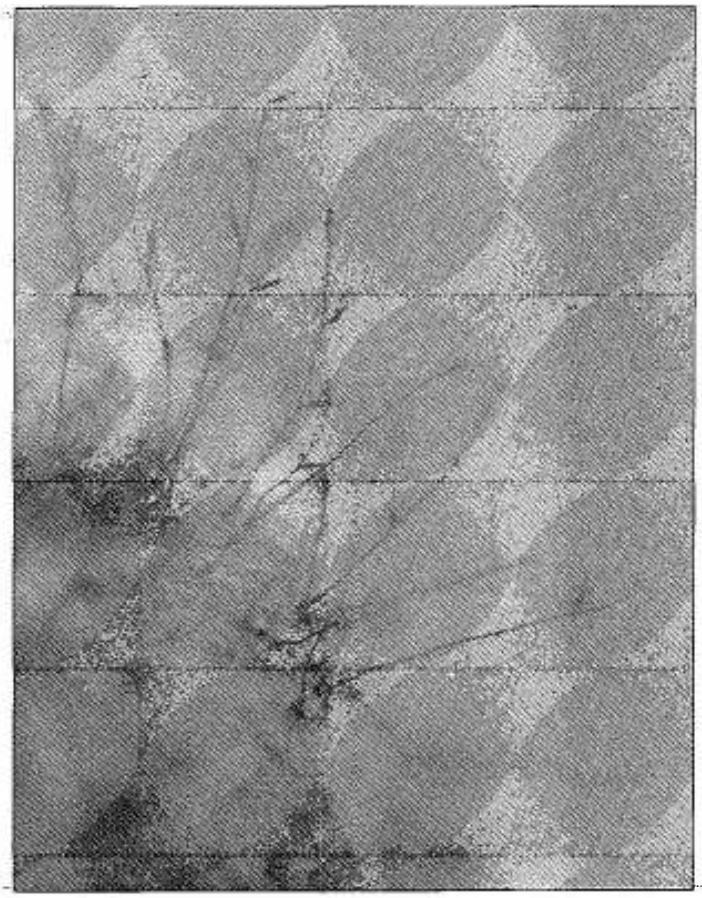


Figure 6B.
Conidiophores and conidia of E. turcicum, *×113.*

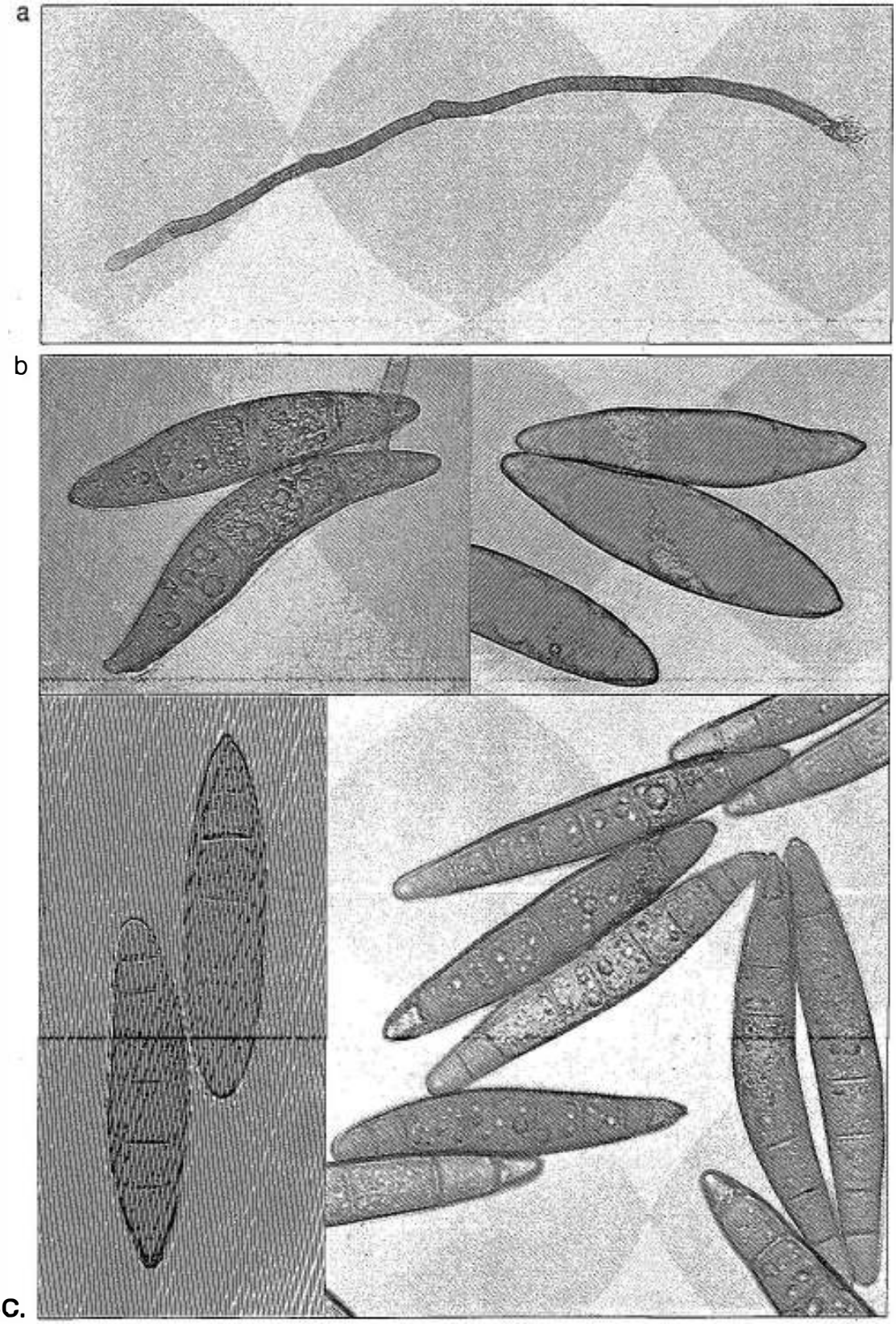


Figure 6C.
(a) Conidiophore, ×451, and (b) conidia of E. turcicum, *×451.*

Fusarium moniliforme J. Sheldon

Fusarium moniliforme causes head blight of sorghum, and twisted top or top rot of pearl millet. Infected seed can be detected by visual examination and incubation tests. A white powdery fungal growth can be seen on dry infected sorghum seed (Fig. 7A). Sometimes, infected seeds of white-seeded sorghum cultivars have a pinkish or violet tinge. Profusely infected seeds are reduced in size and weight, and do not germinate. The fungus usually produces a white to light orange powdery growth consisting of aggregated or loosely scattered chains of microconidia on incubated seed (Fig. 7B). Under the stereobinocular microscope, these microconidial chains appear as fine, thin, long, hairy, white threads (Fig. 7Ca). Sometimes the microconidia may be produced on monophialides in false heads instead of chains (Fig. 7Cb). The **microconidia** are catenulate, hyaline, one- to two-celled, 2–4 × 5–12 µm, and appear as beaded chains (Fig. 7Da). They are oval- to club-shaped with a flattened base (Fig. 7Db). When the microconidia are not produced in chains, they might be confused with those of *F. oxysporum*. However, the phialides are longer and narrower in *F. moniliforme* than in *F. oxysporum*. **Macroconidia** are produced in pale orange sporodochia which can be obscured by the mycelium and the abundant chains of microconidia. Macroconidia are produced on macroconidiophores (Fig. 7Dc). They are hyaline, 3–7 septate, long, 1.5–4 × 20–82 µm in diameter, slender, awl-shaped, falcate to almost straight, and taper towards either end. They are slightly hooked at the tip, thin-walled, with the apical cell slightly curved and tapering to a point, and may be either distinctly or slightly foot-shaped at the basal cell (Fig. 7Dd). Chlamydospores are absent (Ram Nath et al. 1970; Booth 1971; Booth and Waterson 1964; Mathur et al. 1975; Nelson et al. 1983).

Control. Discard infected seeds. Seed treatment with carbendazim or a mixture of benomyl + thiram (Benlate-T®), or carbendazim at about 2 g kg^{-1} is advised to eradicate the seedborne infection (Vidyasekaran 1983).

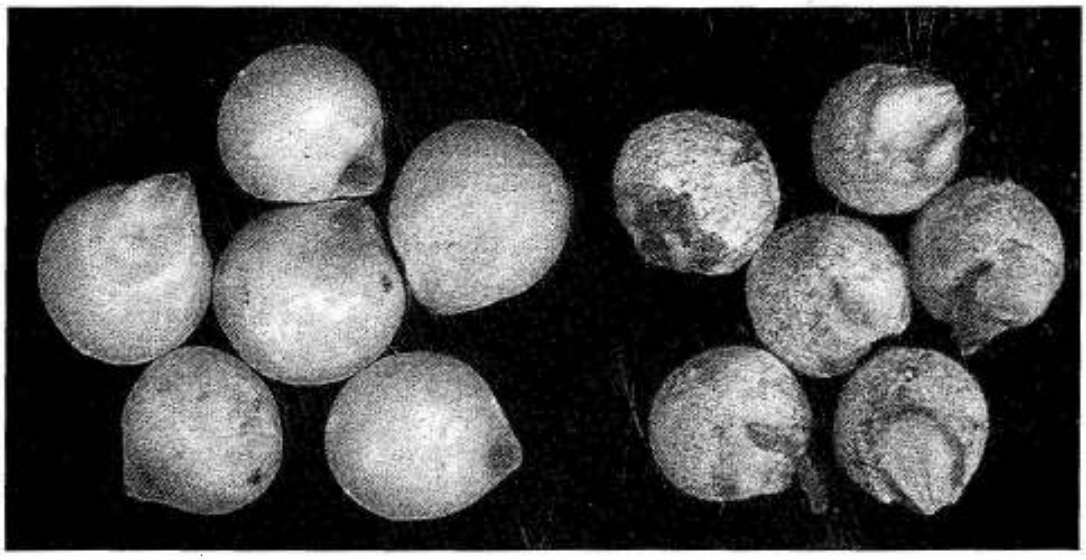


Figure 7A.
Dry infected seed of sorghum showing damage caused by Fusarium moniliforme *(right), and healthy seeds (left), ×11.*

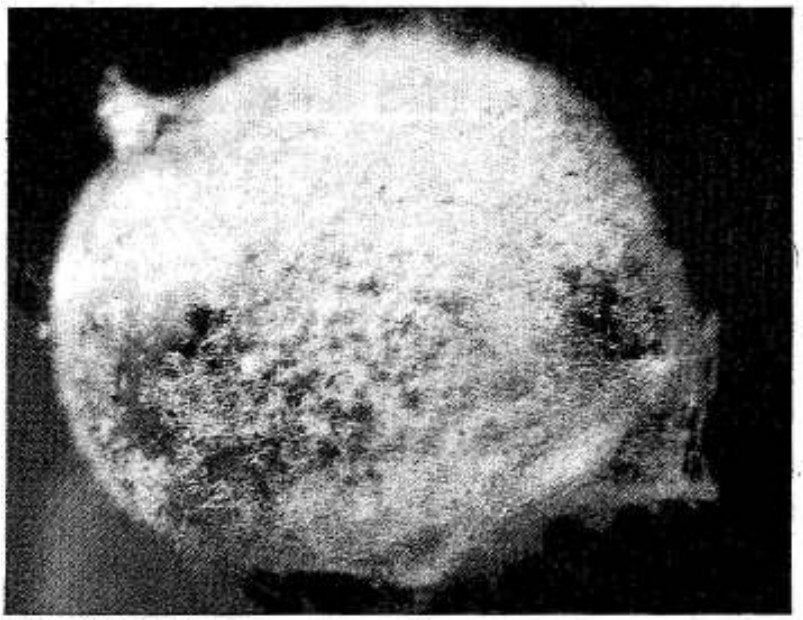


Figure 7B.
Infected sorghum seed after incubation, showing the growth of F. moniliforme, *×22.*

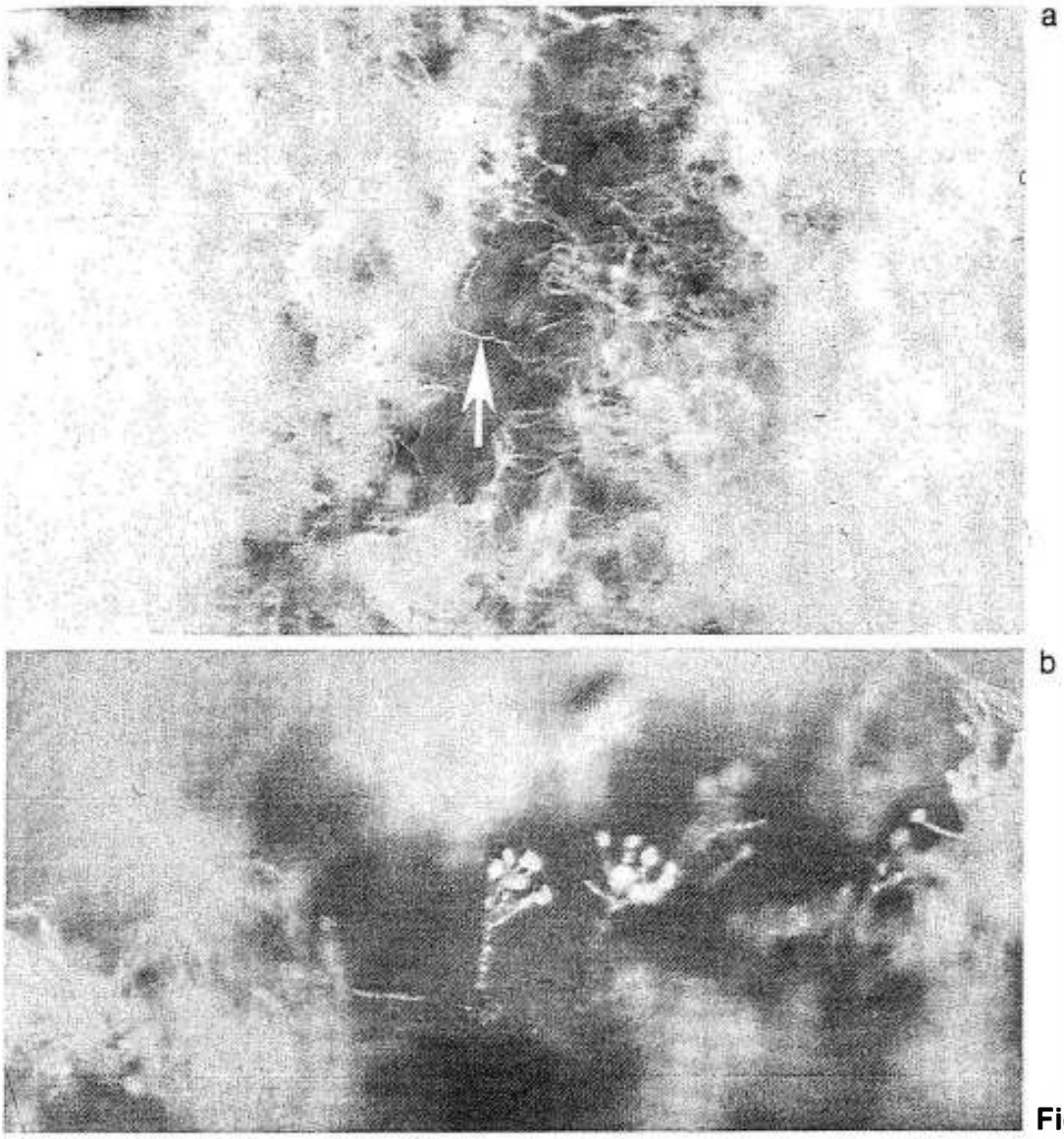


Figure 7C.
(a) Powdery growth containing microconidia in chains, ×122, and (b) dry false heads produced by F. moniliforme*, on sorghum seed in which micro and macroconidia are produced, ×122.*

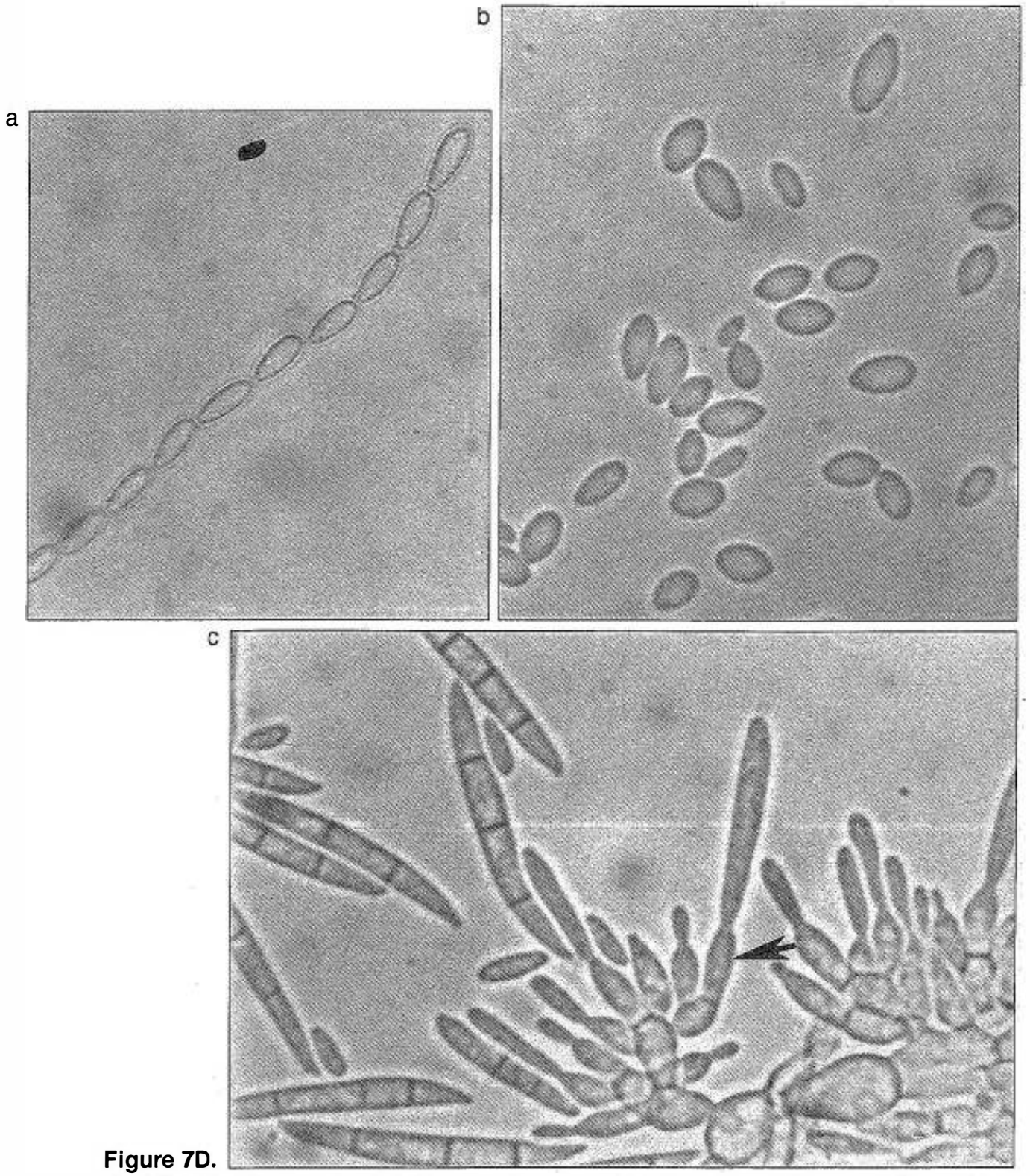


Figure 7D.
(a) Microconidia in beaded chains, ×1130, (b) oval- to club-shaped microconidia, ×1130, (c) macroconidiophores (arrowed), ×1130, and

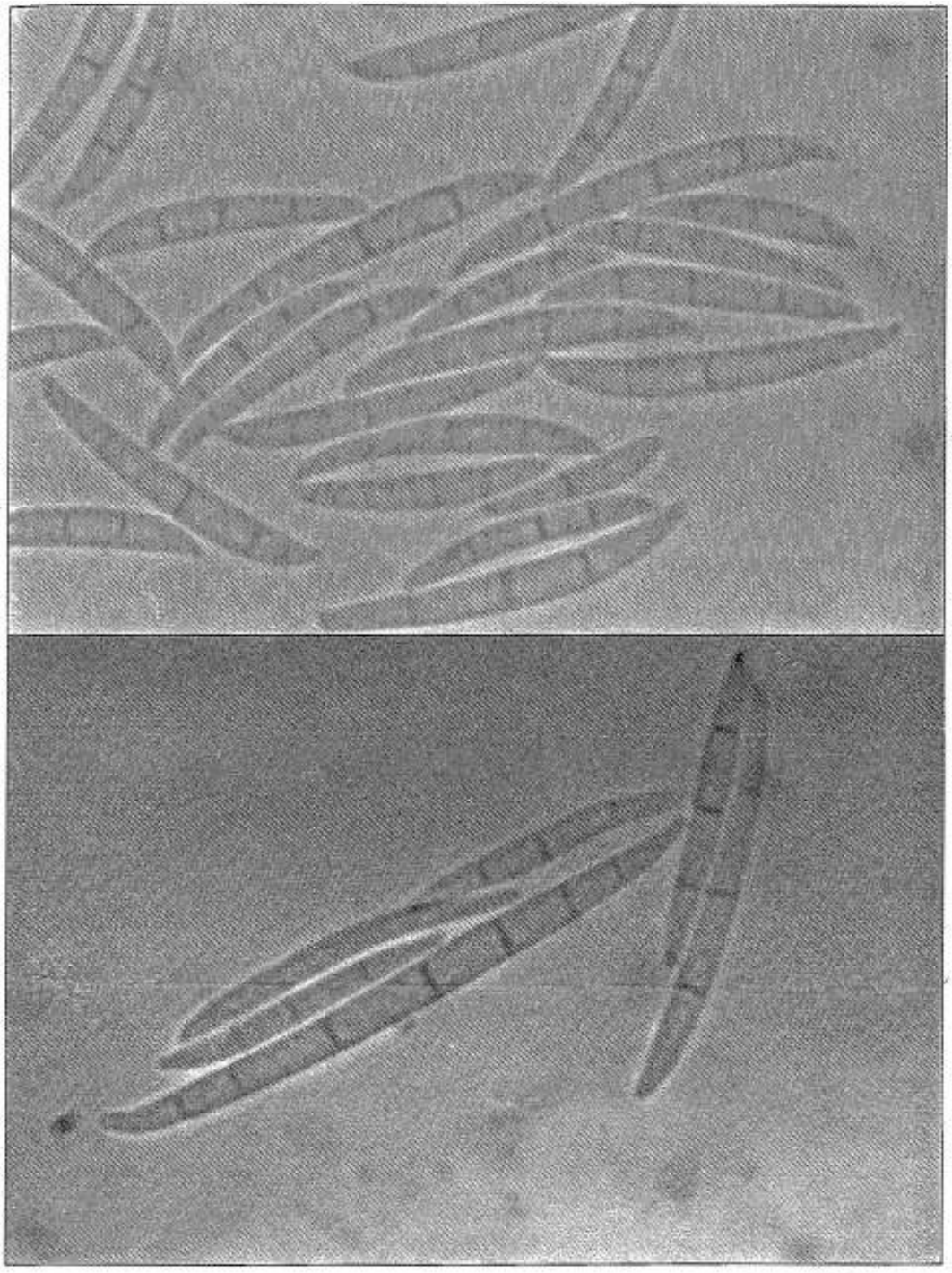


Figure 7D.
(d) macroconidia of F. moniliforme, *×1130.*

Gloeocercospora sorghi Bain and Edgerton ex Deighton

Gloeocercospora sorghi causes zonate leaf spot of sorghum. Seeds carry the fungus as both sclerotia and dormant mycelium. Infected seeds can be detected by visual examination and incubation tests. Dry infected seeds show black, shiny, spindle- or irregular-shaped sclerotia, about 0.1–0.2 mm in diameter (Fig. 8A). The sclerotia are embedded in the seed coat, and often become erumpent by rupturing it. On incubated seed, the fungus produces dark brown to charcoal black sclerotia, and pink to reddish orange sporodochia (Fig. 8B). Sometimes only sclerotia are produced. The **mycelium** is scanty or abundant, white to dull white, thin and branched. **Sporodochia** are pink to salmon pink, and are visible to the naked eye. Each sporodochium consists of numerous hyaline conidiophores and conidia, that can be seen under a compound microscope. **Conidiophores** are hyaline, branched or unbranched, septate, short, 5–10 µm long, with a somewhat swollen apex. **Conidia** are borne in a pinkish, slimy matrix, and are hyaline, elongate to filiform, 1.4–3.2 × 20–195 µm, and septate (Fig. 8Ca and b). **Sclerotia** on incubated seed appear similar to those produced on dry seed (Mulder and Holliday 1971; Watanabe and Hashimoto 1978).

Control. Collect seed from disease-free fields. Discard infected seed lots particularly those originating from countries where the disease occurs. Seed dressing with carbendazim (Bavistin® at about 1 g kg^{-1}) or thiabendazole (Tecto-50® at about 2 g kg^{-1} of seed) is effective in reducing the seedborne inoculum (Mathur et al. 1987).

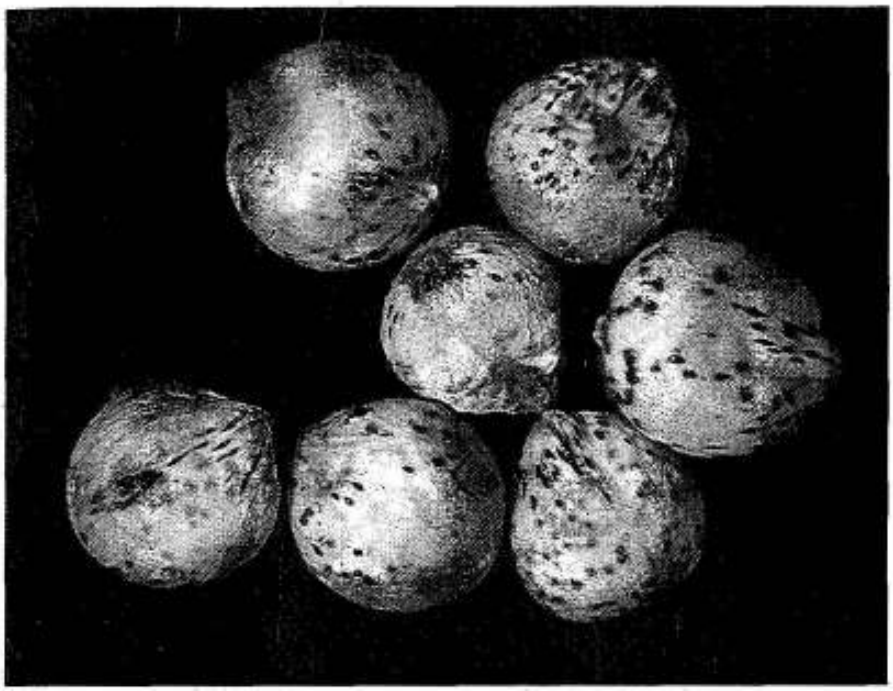


Figure 8A.
Dry infected seeds of sorghum showing damage caused by Gloeocercospora sorghi, *×11.*

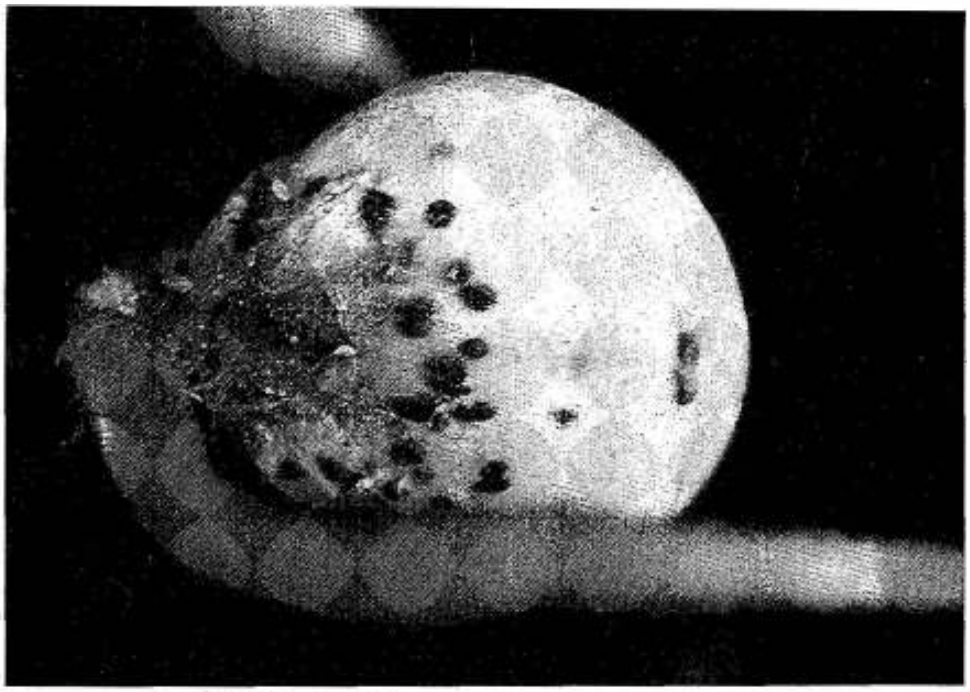


Figure 8B.
Infected sorghum seed after incubation, showing the growth of G. sorghi, *×27.*

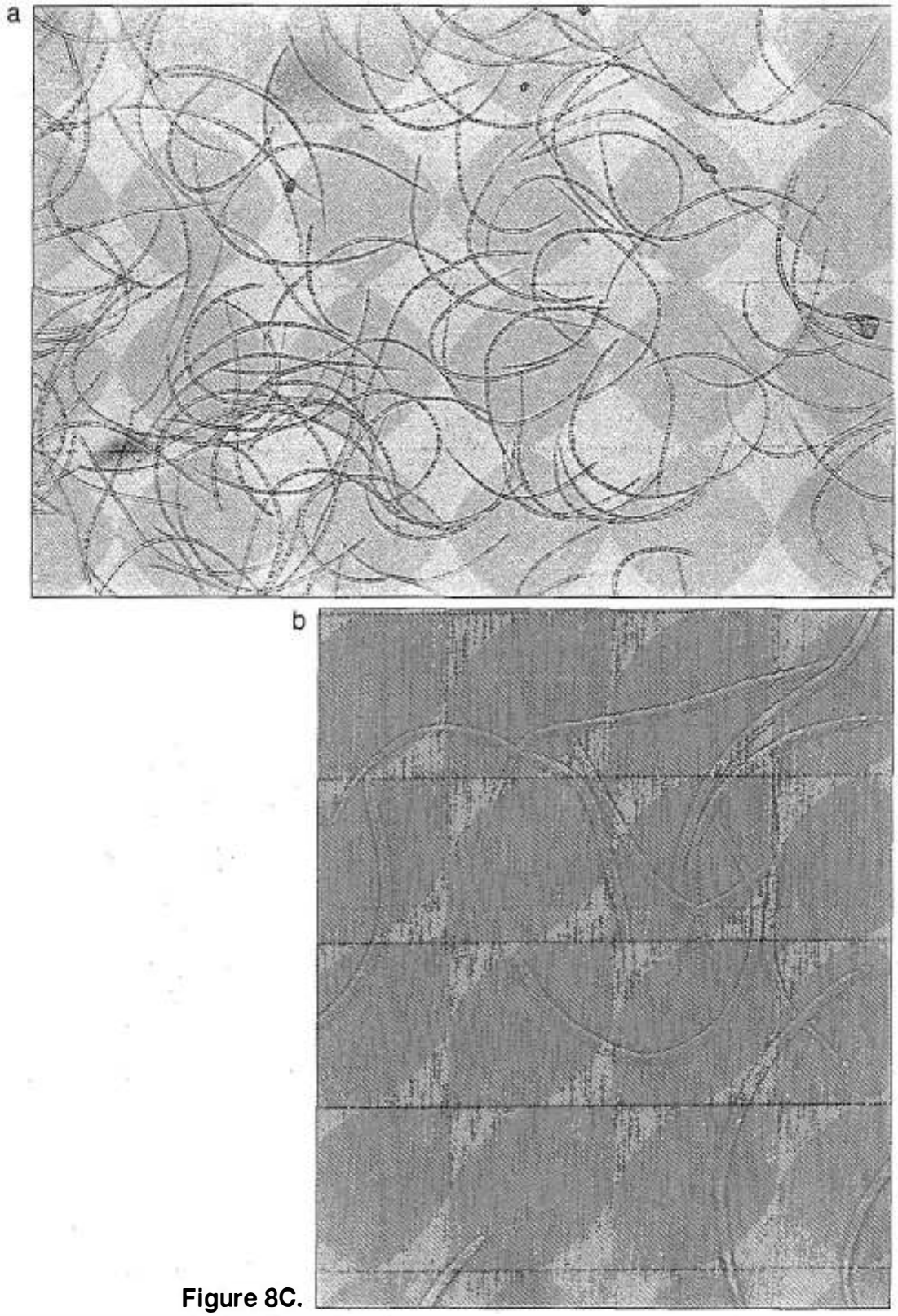


Figure 8C.
(a) Hyaline, elongate to filiform conidia, ×225, and (b) conidia of G. sorghi, *×451.*

Peronosclerospora sorghi (W. Weston & Uppal) C.G. Shaw

Sclerospora sorghi W. Weston & Uppal

This fungus is an obligate parasite, and causes downy mildew of sorghum. Seeds harvested from diseased fields are likely to carry the fungus in the form of oospores adhering to their surface, and as dormant mycelium present in internal seed tissue. Seeds carrying oospores can be detected by a seed-washing test (see Appendix 2). The **oospores** are smooth, globose, spherical, hyaline, yellow, and 25–43 µm in diameter. Oogonial walls are irregularly thick, and are yellow. The contents of the oospores are finely granular and rich in oil globules (Fig. 9) (Safeeulla 1976; Francis and Williams 1983b).

Control. The introduction of this disease into new areas can be prevented by: conducting pre-export crop health inspections, producing seed from a field certified to be free from downy mildew, and distributing seed of cultivars with a high degree of resistance to sorghum downy mildew.

These measures eliminate the possibility of oospore-contaminated seed being exported without recognition.

Dressing seed with systemic fungicides such as metalaxyl at about 1 g kg^{-1} eradicates the seedborne inoculum (Anahosur and Patil 1980).

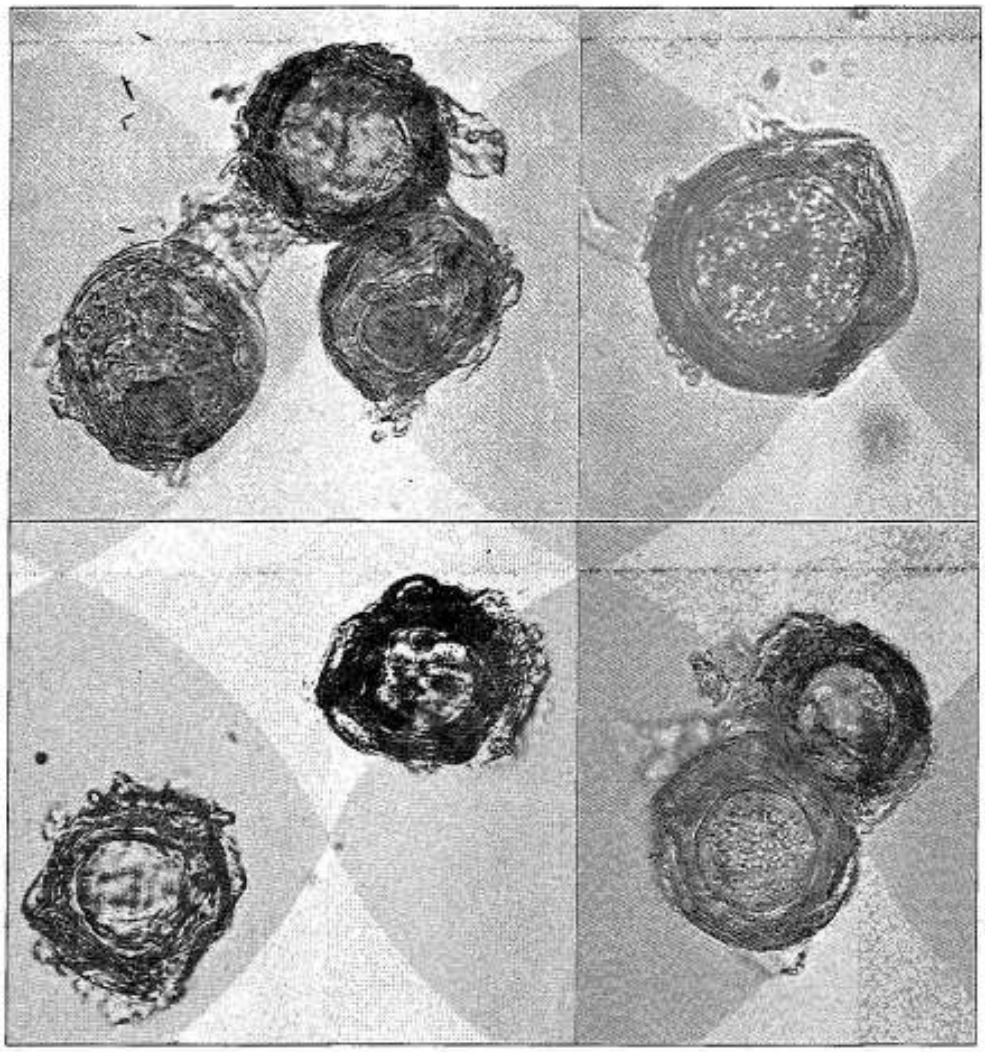


Figure 9.
Oospores of Peronosclerospora sorghi, *×451.*

Phoma sorghina (Sacc.) Boerema, Dorenbosch, & van Kesteren

Phoma insidiosa Tassi

Phoma sorghina causes leaf spot and blight of sorghum. The seedborne inoculum of the fungus causes considerable damage. The fungus is carried as pycnidia and as dormant mycelium. Infected seeds can be detected by visual examination and incubation tests. Numerous pycnidia can be seen on dry seeds under a magnifying lens (Fig. 10A). The pycnidia are dark brown to black and the size of a pinhead. The fungus produces black pycnidia scattered throughout the surface of incubated seed. When the seed is heavily infected, the fungus can rupture the seed coat giving seed a cancerous or warty appearance (Fig. 10Ba). Sometimes fungal growth on incubated seed consists only of mycelium and chlamydospores (Figs. 10Bb and 10Ca). The **mycelium** is profuse, fluffy to dense, and is often very variable in color. Sometimes pycnidia are produced on the aerial mycelium. **Pycnidia** may be immersed or erumpent, dark brown to black, shiny or dull (Fig. 10Cb). They are single or multiostiolate, variable in shape, often globose to subglobose, and 60–150 µm in diameter. The beak is very small or inconspicuous, and cirrhus is not usually produced. **Conidia** are hyaline, single-celled, variable in shape, usually globose to ovoid or shortly cylindrical, they measure 1.4–4.4 × 3.5–8.8 µm in diameter, and are straight and guttulate (Fig. 10Da). **Chlamydospores** are frequently produced on the aerial mycelium and directly on the seed surface in a catenuliform. They are single- to several-celled, dark, thick-walled, resemble *Alternaria* spores, and are sometimes irregular in shape (Fig. 10Db) (Tarr 1962; Punithalingam 1985; Sutton 1980).

Control. Control measures should include selection of seed from noninfected plants combined with seed treatment. During visual examination infected seeds should be discarded. Seed treatment with thiram, captan or mancozeb (Dithane M-45®) fungicides at about 3 g kg^{-1} is advised to reduce the seedborne inoculum (Anahosur and Hegde 1980).

Figure 10A.
Dry seeds of sorghum showing damage caused by Phoma sorghina, ×*12.*

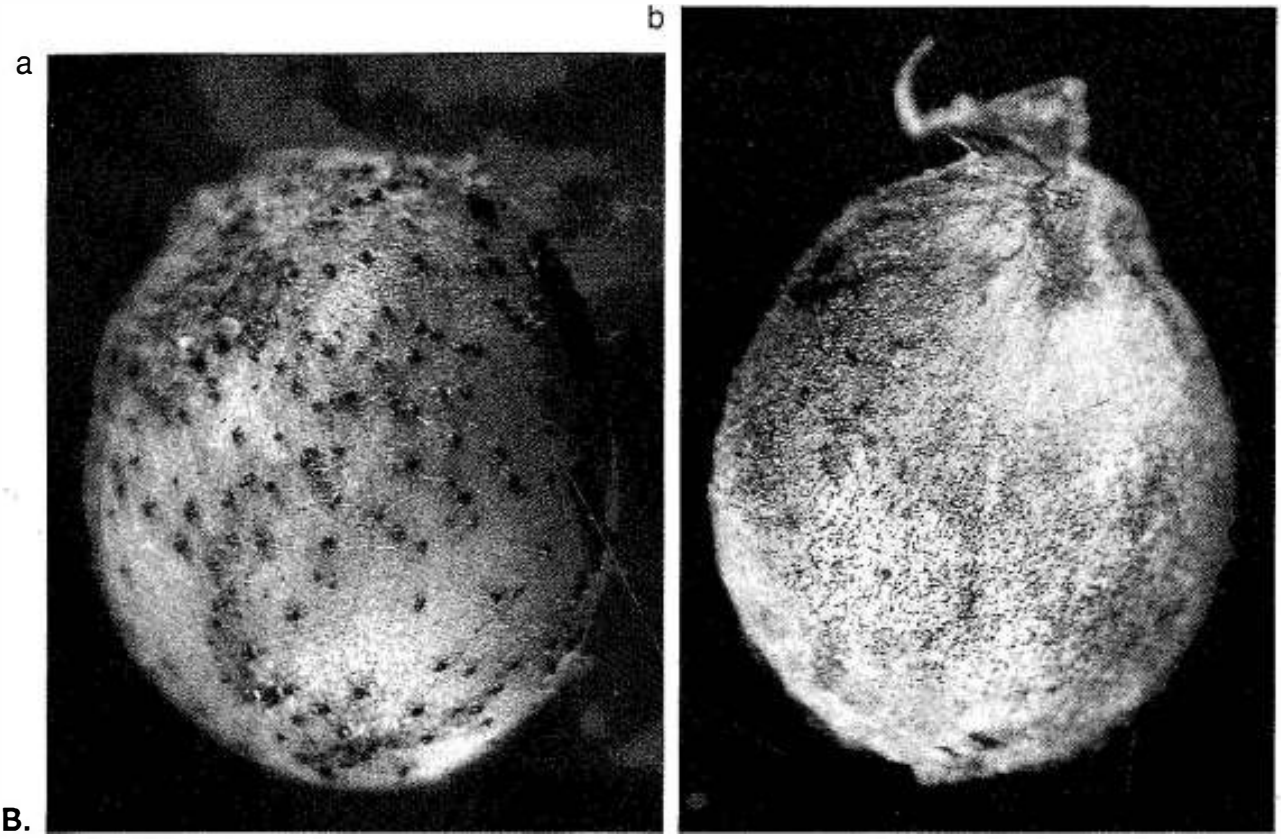


Figure 10B.
Infected sorghum seed after incubation, showing two types of fungal growth; (a) pycnidial, ×*27, and (b) chlamydospores of* P. sorghina, ×*24.*

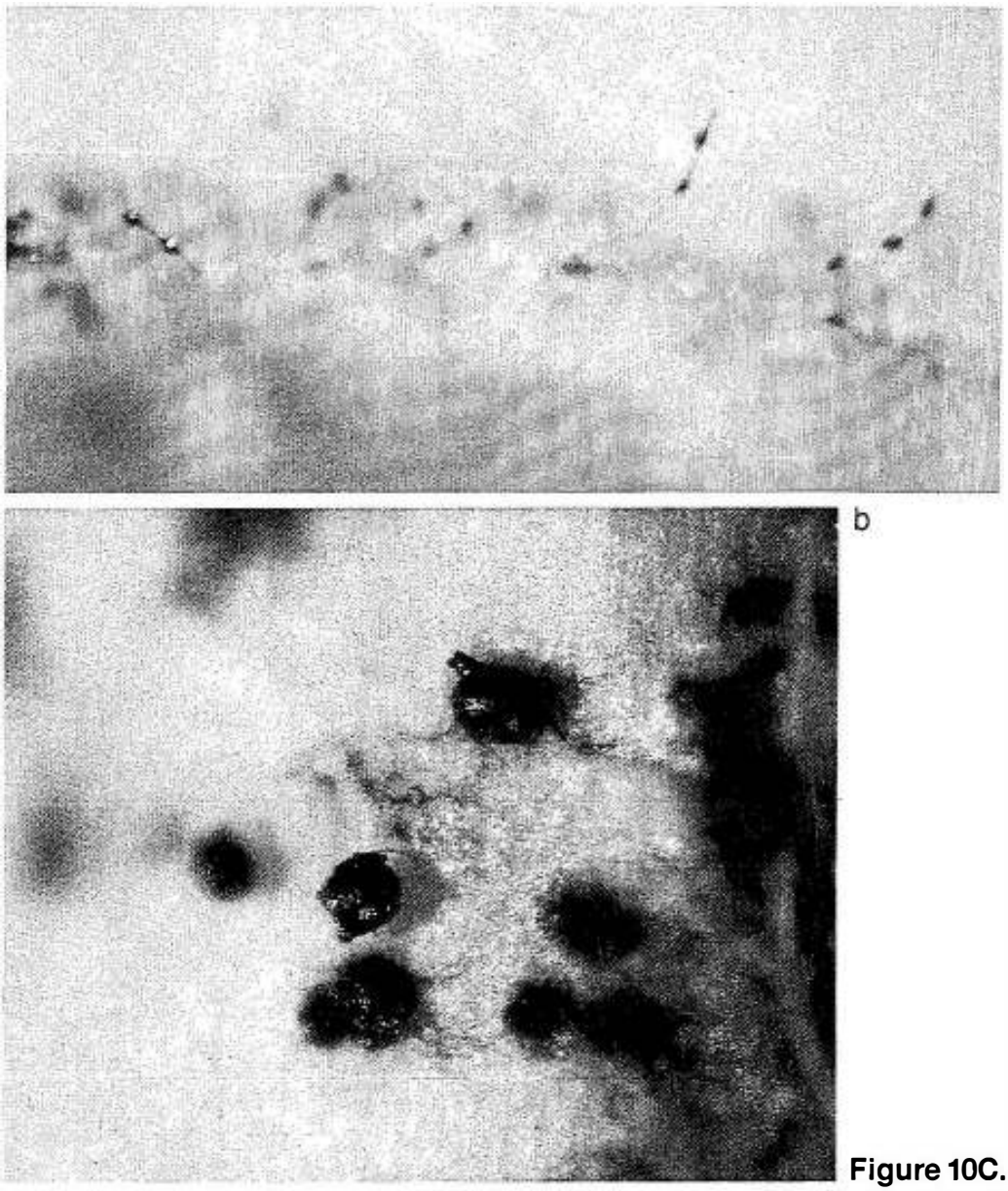


Figure 10C.
(a) Chlamydospores, ×113, and (b) pycnidia of P. sorghina, *×113.*

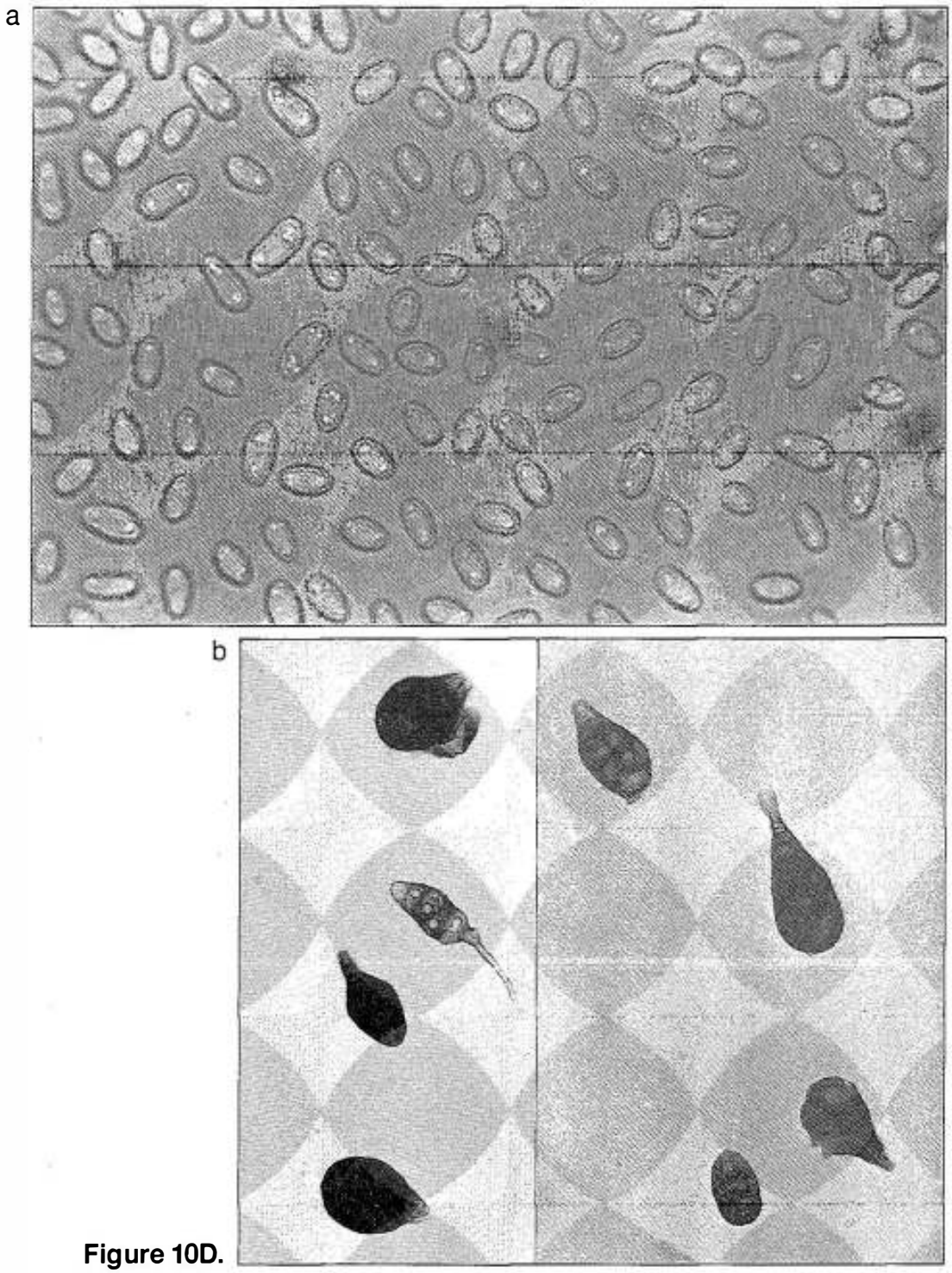


Figure 10D.
(a) Conidia, ×1130, and (b) chlamydospores of P. sorghina, *×225.*

Sporisorium cruentum (Kühn) K. Vánky

Sphacelotheca cruenta (Kühn) A.A. Potter

Sporisorium cruentum causes loose smut of sorghum. The fungus is seedborne as chlamydospores. During crop maturation, harvesting, and threshing, grain from fields containing smutted plants becomes contaminated with chlamydospores. Chlamydospores adhering to healthy-looking seed can be detected by seed-washing test, they can be identified under the compound microscope by their shape and color. The **chlamydospores** are globose to subglobose, elliptical or slightly irregular in shape, light yellowish-brown to dark olivaceous brown, and finely echinulated (Fig. 11). They are slightly darker and larger than the spores of *Sporisorium sorghi*, and range in diameter from 5 to 10 µm, 1-µm

Control. Conduct pre-export crop health inspections during the growing season. Seed treatment with carboxin at about 2.5 g kg^{-1} can be used to control the smut fungi. Elemental sulfur at about 5 g kg^{-1} is also effective in reducing the seedborne inoculum (Neergaard 1979).

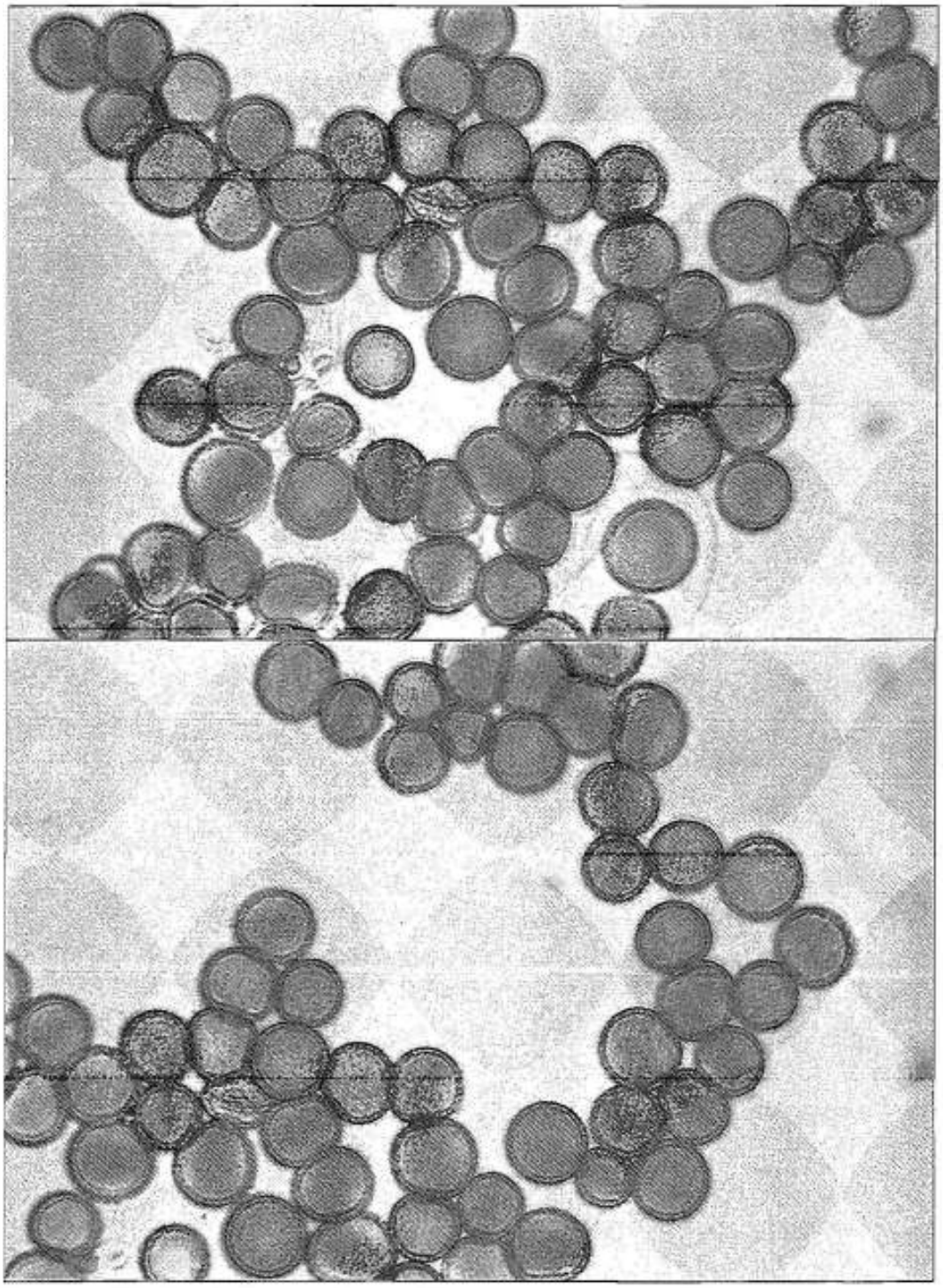


Figure 11.
Chlamydospores of Sporisorium cruentum, *×1130.*

Sporisorium holci-sorghi (Rivolta) K. Vánky

Sporisorium reilianum (Kühn) McAlpine

Sphacelotheca reiliana (Kühn) G.P. Clinton

This species causes head smut of sorghum and maize. Infected plants produce smutted earheads. The surfaces of seeds harvested from diseased fields are likely to be contaminated with chlamydospores of the fungus, which can be detected by a seed-washing test. Under the compound microscope, these **chlamydospores** appear somewhat opaque, dark brown, globose to subglobose. They are usually somewhat angular, minutely, but abundantly papillate, 9–14 µm in diameter, and their spore walls are about 1 µm thick (Fig.12). Sometimes sterile hyaline cells intermixed with chlamydospores can be seen (Tarr 1962; Ainsworth 1965b).

Control. Avoid collecting seeds from endemic fields. Seed treatment with carboxin at about 2.5 g kg^{-1} or elemental sulfur at about 5 g kg^{-1} helps to eradicate the seedborne inoculum (Neergaard 1979).

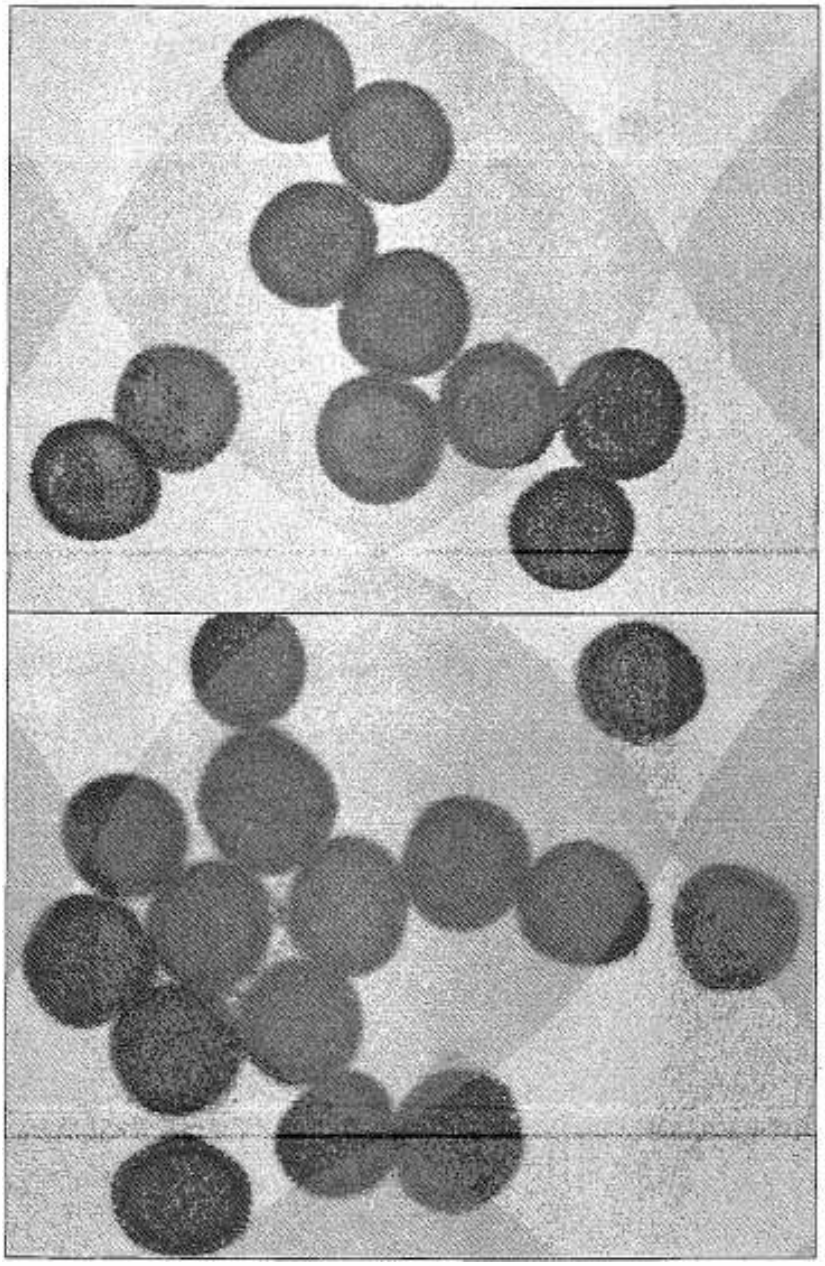


Figure 12.
Chlamydospores of Sporisorium holci-sorghi, *×1130.*

Sporisorium sorghi Link

Sphacelotheca sorghi (Link) G.P. Clinton

This fungus causes grain smut of sorghum. Its seedborne inoculum can be carried as admixtures of smut sori, and as chlamydospores adhering to the seed surface. Smut sori are visible to the naked eye, and can be identified under a magnifying lens with a magnification of ×2, by their shape, size, and color (Fig. 13A). Chlamydospores present on contaminated seed can be detected by a seed-washing test. Smut **sori** are oval, cylindrical, or nearly conical in shape, sometimes surrounded by unaltered glumes at the base. In some sorghum varieties, the shape of the smut sori is similar to that of normal grain, but when such grains are broken, they reveal the dark powdery spores of the fungus. Smut sori are between 4.32–12.7 mm wide and 4.23–38.1 mm long. They vary considerably in size, but even the longest are much shorter than those of long smut. The **chlamydospores** of this fungus are very similar to those of *S. cruentum*, but are usually slightly smaller and somewhat paler brown. They are globose to subglobose, and appear smooth, but under high magnification, minute echinulations can be seen (Fig. 13B). Individual spores range between 3–9 μm in diameter (Tarr 1962; Ainsworth 1965c).

Control. Avoid collecting seed from endemic areas. Discard smut sori during visual examination. Treat apparently healthy-looking seed with carboxin (Vitavax® at about 2 g kg^{-1}) (Atac 1989), or elemental sulfur at about 5 g kg^{-1} (Neergaard 1979). If chemical seed dressings cannot be obtained, soak the seeds in water for 4 h, then dry them, first in the shade and then in the sun. This procedure kills germinating smut spores without impairing seed viability (Doggett 1988).

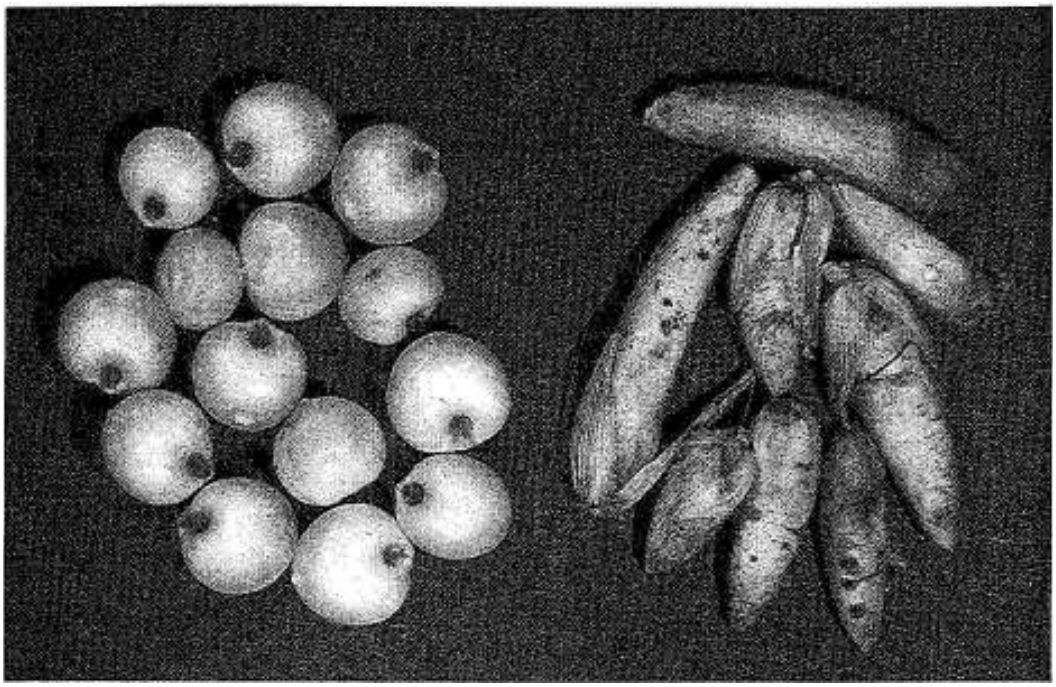


Figure 13A. *Smut sori produced by* Sporisorium sorghi *(right), and healthy sorghum seed (left), ×11.*

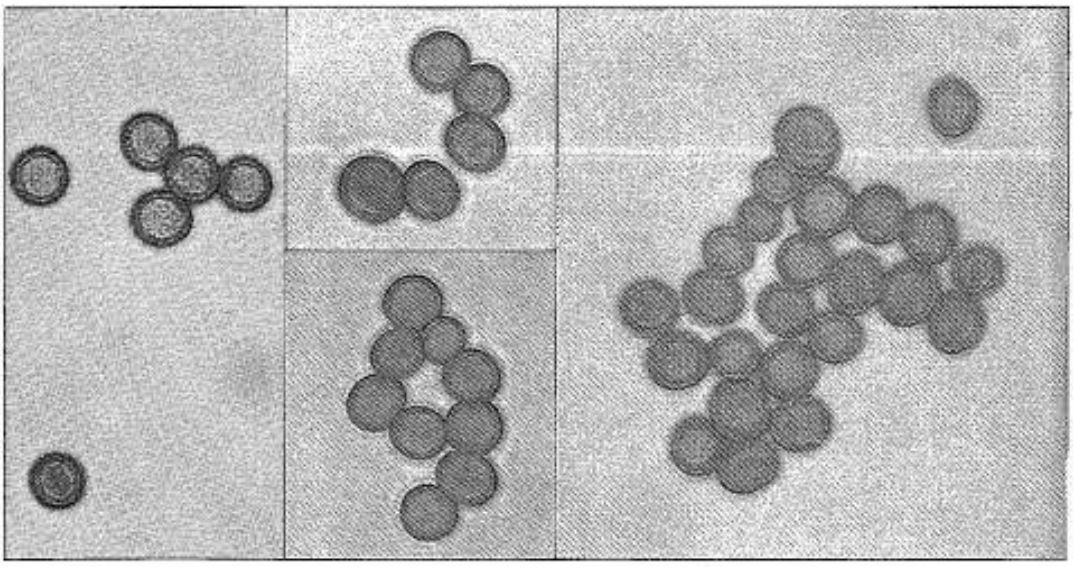


Figure 13B. *Chlamydospores of* S. sorghi *showing minute echinulations, ×1130.*

Tolyposporium ehrenbergii (Kühn) Patouillard

This fungus causes long smut of sorghum. The sori produced by *Tolyposporium ehrenbergii* can be detected in sorghum seed lots as admixtures during visual examination. In addition to smut sori, healthy-looking seed might also be contaminated by teliospores (chlamydospores) adhering to its surface. Teliospores can be detected by a seed-washing test. Long smut sori which are visible to the naked eye, are cylindrical, elongate, and slightly curved (Fig. 14A). They are considerably longer and wider than those of covered smut *Sporisorium sorghi*, and are 4.23–12.7 mm in diameter, and 25.4–63 mm long. Smut sori consist of masses of spore balls enclosed in a thick light yellowish-brown membrane which splits irregularly from the apex downwards to reveal a dark, solid mass of spores. The spore balls are light brownish-yellow, 45–200 µm in diameter, and consist of a mass of teliospores. **Teliospores** are single-celled, brown and opaque, globose to oval or irregular in shape. The spores towards the periphery of the spore ball are dark brown and echinulate, with papillate on the free surface. The spores towards the center of the spore ball are light brown, and have a smooth surface. They are usually globose, but somewhat angular, ovate, flattened, brownish-green, and 10–15 µm in diameter (Fig. 14B) (Tarr 1962; Ainsworth 1965d).

Control. Conduct pre-export crop health inspections to select disease-free seed. Discard smut sori during visual examination. Chemical seed dressing with thiram at about 2 g kg^{-1} is effective in reducing the seedborne inoculum (Kumar and Viswanath 1991).

Figure 14A.
Smut sori produced by Tolyposporium ehrenbergii *in sorghum (right), and healthy seeds (left), ×11.*

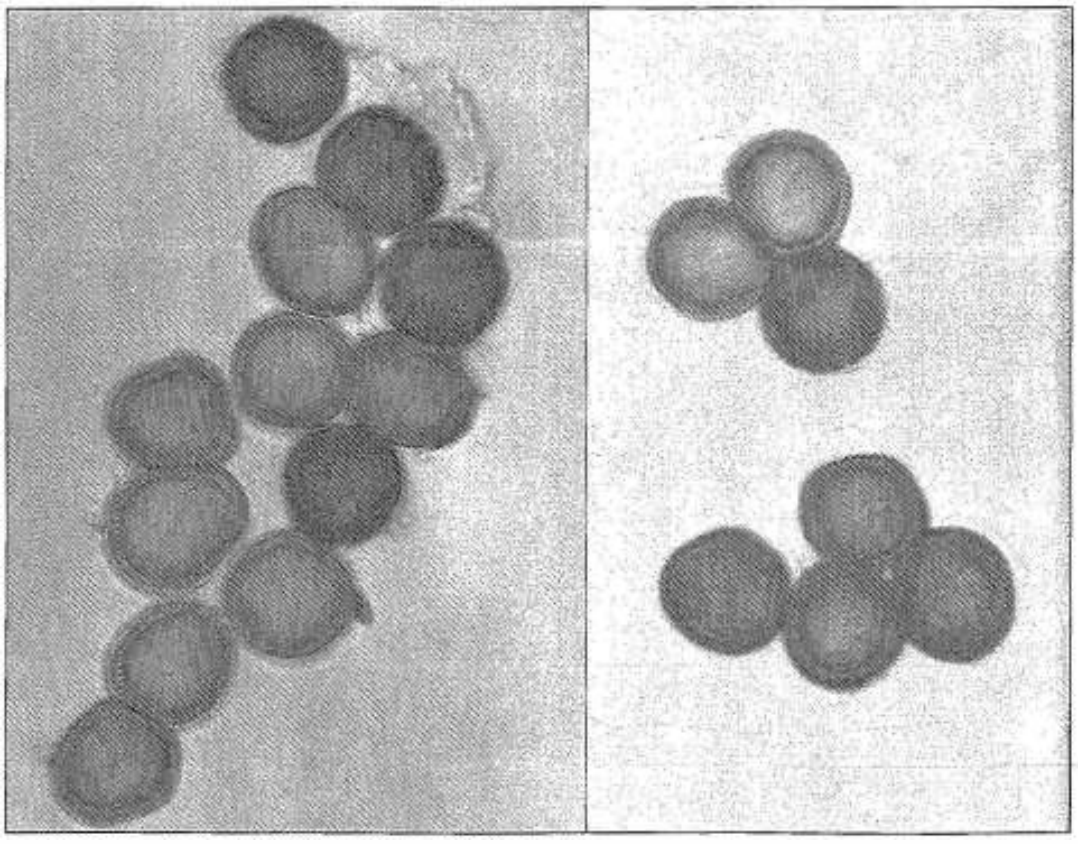


Figure 14B.
Teliospores of T. ehrenbergii, *×1130.*

Pearl Millet

Bipolaris sacchari (E.J. Butler) Shoemaker

Helminthosporium sacchari E.J. Butler

Drechslera sacchari (E.J. Butler) Subramanian & P.C. Jain

This fungus causes eye spot and seedling blight of pearl millet under very humid conditions. It is known to occur particularly in areas where sugarcane is grown. Infected pearl millet seeds can be detected by incubation tests (Fig.15A). On incubated seed, the fungus produces light brown conidiophores on which long, thin, slightly curved conidia are borne in various ways (Fig.15B). The **mycelium** is similar in morphology to that of any other *Bipolaris* species. The **conidiophores** arise singly or in groups of two to four, have a swollen base, and are long, thin, straight or flexuous, olivaceous brown, paler towards the apex, septate, smooth, 200 µm long, 5–8 µm thick, and bear conidia at short intervals. **Conidia** are 5–9 distoseptate, mid-pale to mid-yellow, golden brown, solitary, acropleurogenous, usually straight but sometimes slightly curved, long, cylindrical, and measure 35–96 × 9–17 µm (Fig.15C). They have a broad and distinct hilum which lies within the contour of the basal cell (Ellis 1971; Ellis and Holliday 1971b; Chidambaram et al. 1973; Misra et al. 1974; Sivanesan 1987).

Control. Seed treatment with copper oxychloride + zineb (Miltox® at about 2.5 g kg^{-1}) is advised to eradicate the seedborne inoculum (Grower and Suryanarayana 1970).

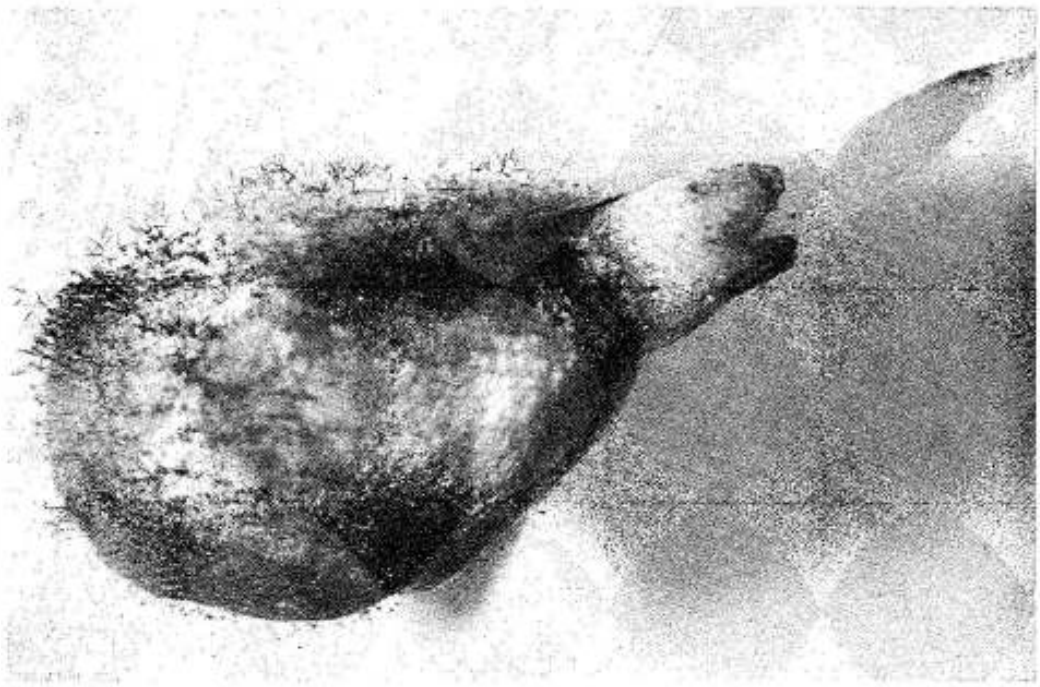


Figure 15A.
Infected pearl millet seed after incubation, showing the growth of Bipolaris sacchari, ×*40.*

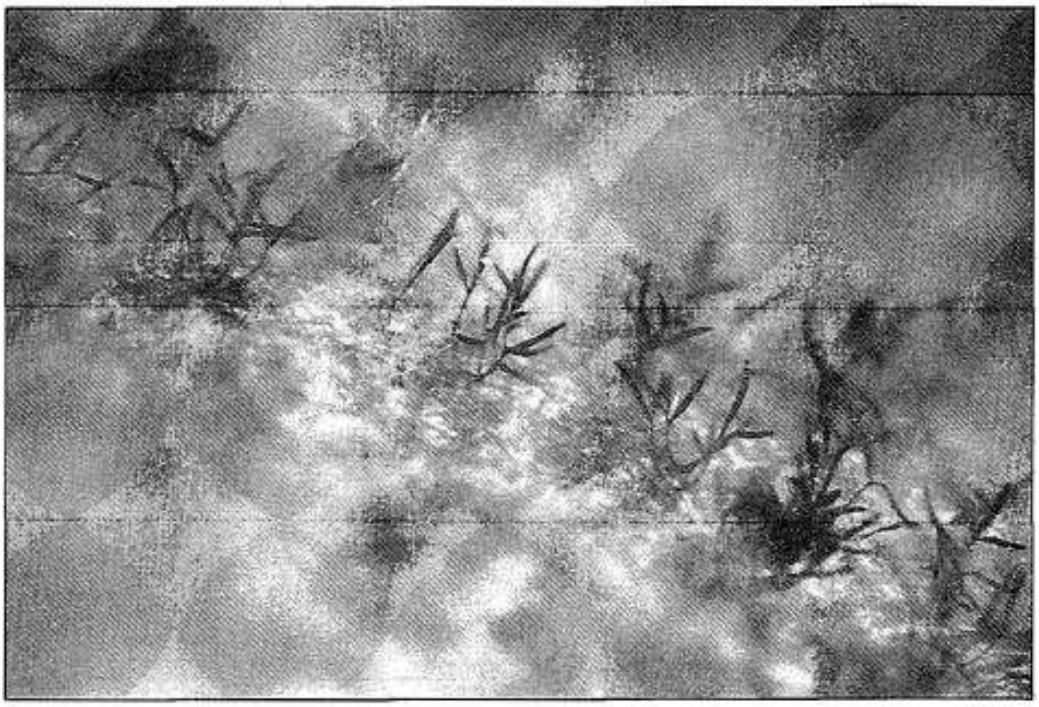


Figure 15B.
Conidiophores and conidia of B. sacchari, ×*127.*

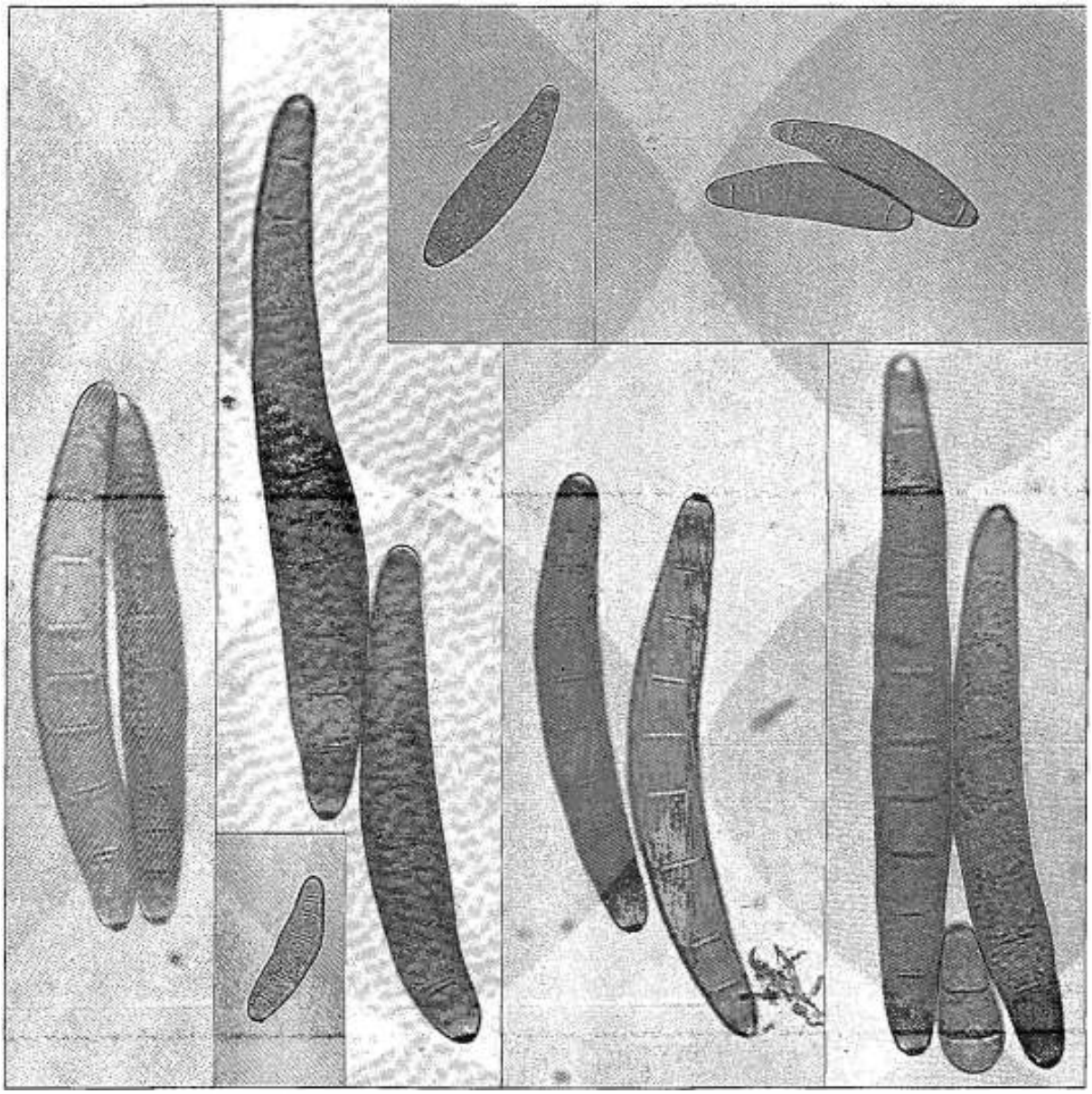


Figure 15C.
Conidia produced by B. sacchari, *×677.*

Bipolaris setariae (Sawada) Shoemaker

Helminthosporium setariae Sawada

Drechslera setariae (Sawada) Subramanian & P C Jain

Cochliobolus setariae (Ito & Kuribayashi) Drechs ex Dastur

This fungus causes leaf spots of pearl millet and finger millet, and is known to be seedborne. Infected seeds can be detected by visual examination and incubation tests. Dry infected seed sometimes appears brown and more intensely colored than normal pearl millet seed (Fig.16A) On incubated seed, the fungus produces a hairy or velvety, olive brown to black growth (Figs.16Ba and b). Sometimes a perfect state of the fungus is also produced on infected seed, when this happens dark-colored perithecia can be seen growing either superficially or partly embedded in the seed coat (Fig.16Bc). The **mycelium** is dark, individual hyphae are irregularly branched with rough surfaces. **Conidiophores** arise singly or in small groups throughout the surface, and are up to 200 µm long and 5–9 µm wide. They are simple, erect, straight, or sometimes slightly curved, and dark brown to black. The color becomes paler towards the apical portion of the conidiophore, while the base remains dark and swollen (Figs. 16Ca and 16Da). **Conidia** are 5–10 distoseptate, pale to mid-golden brown, solitary, acrogenous, acropleurogenous, cylindrical, slightly curved or rarely straight, fusiform or navicular, and 45–100 × 10–15 µm (Fig.16Db). Secondary conidia are produced on the conidiophores growing from the primary conidia, but not so frequently as in *B. sorghicola*. **Perithecia** develop within the stroma and are dark brown to black, unilocular with a globose body and a paraboloid to cylindric ostiolar neck, measuring 240–500 × 220–350 µm. Often sterile, hyaline, brown hyphae cover the perithecia (Fig.16Cb). The perithecia contain **asci** which are vestigial, bitunicate, hyaline, cylindrical to obclavate, short, often becoming more or less distended especially before dehiscence, and 130–150 × 22–32 µm (Fig.16Dc and Dd). Each ascus contains eight **ascospores** (Fig.16De) which are hyaline, indistinctly pluriseptate, coiled, and filiform. Ascospores are helicoid, 5–9 septate, 200–315 × 6–7 µm and are violently discharged (Wells and Winstead 1965; Ellis and Gibson 1975; Shetty et al. 1982; Sivanesan 1987).

Control. Conduct pre-export crop health inspections. Discard infected seeds during visual examination. Seed dress pearl millet with benzothiazole (Delsan-30® at about 1 mL kg^{-1}) to eradicate the seedborne inoculum (Rao et al. 1984).

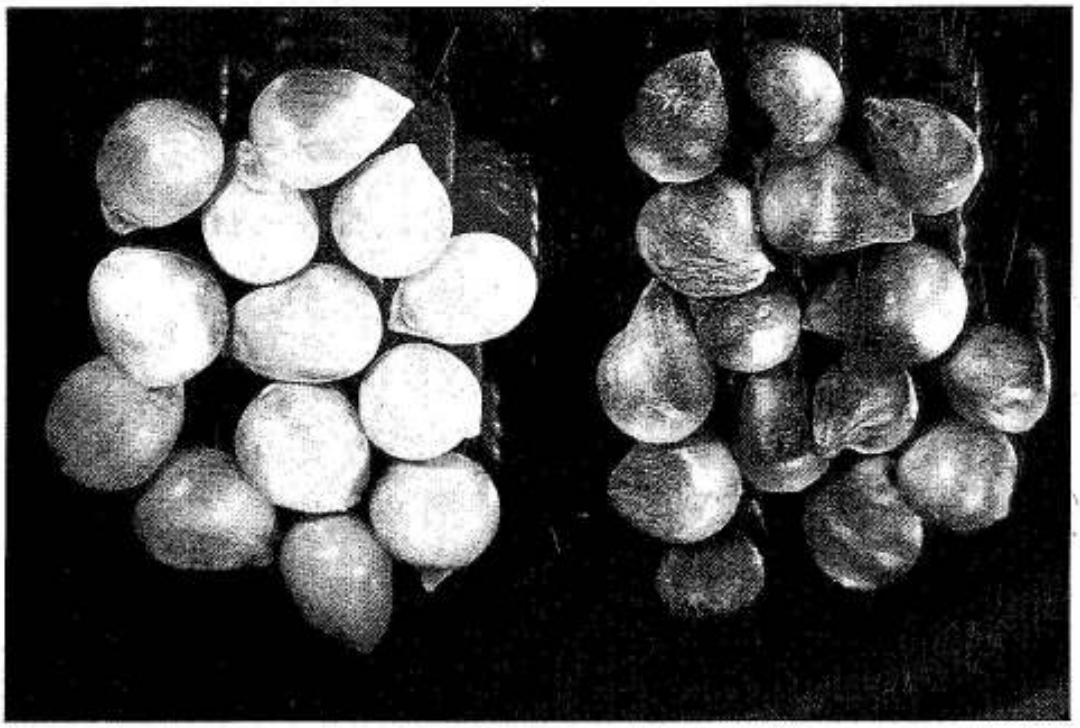


Figure 16A.
Dry, deep brown seeds of pearl millet showing damage caused by Bipolaris setariae *(right), and healthy seed (left), ×11.*

a

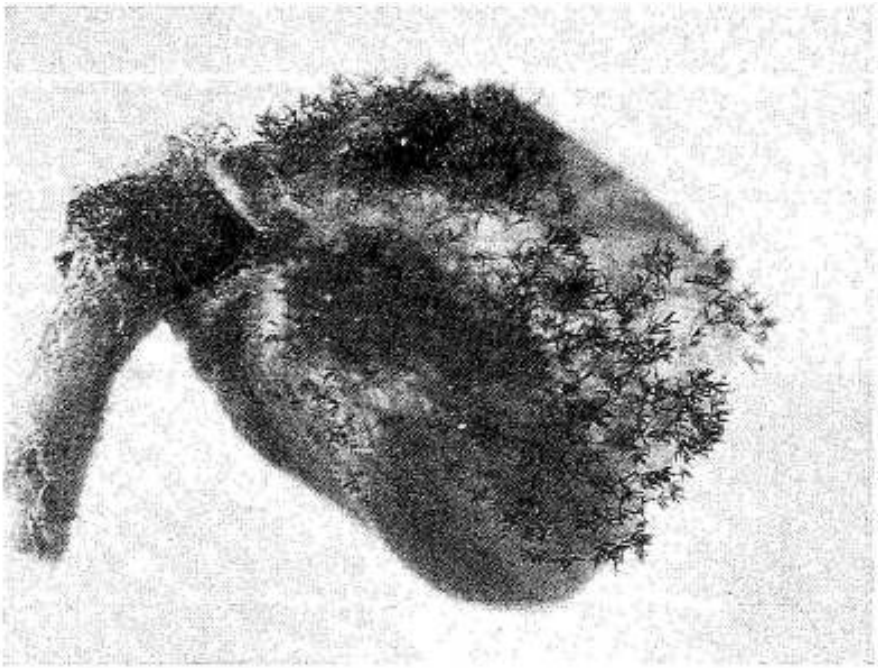


Figure 16B.
(a) Hairy, velvety B. setariae *fungus on incubated pearl millet seed, ×40,*

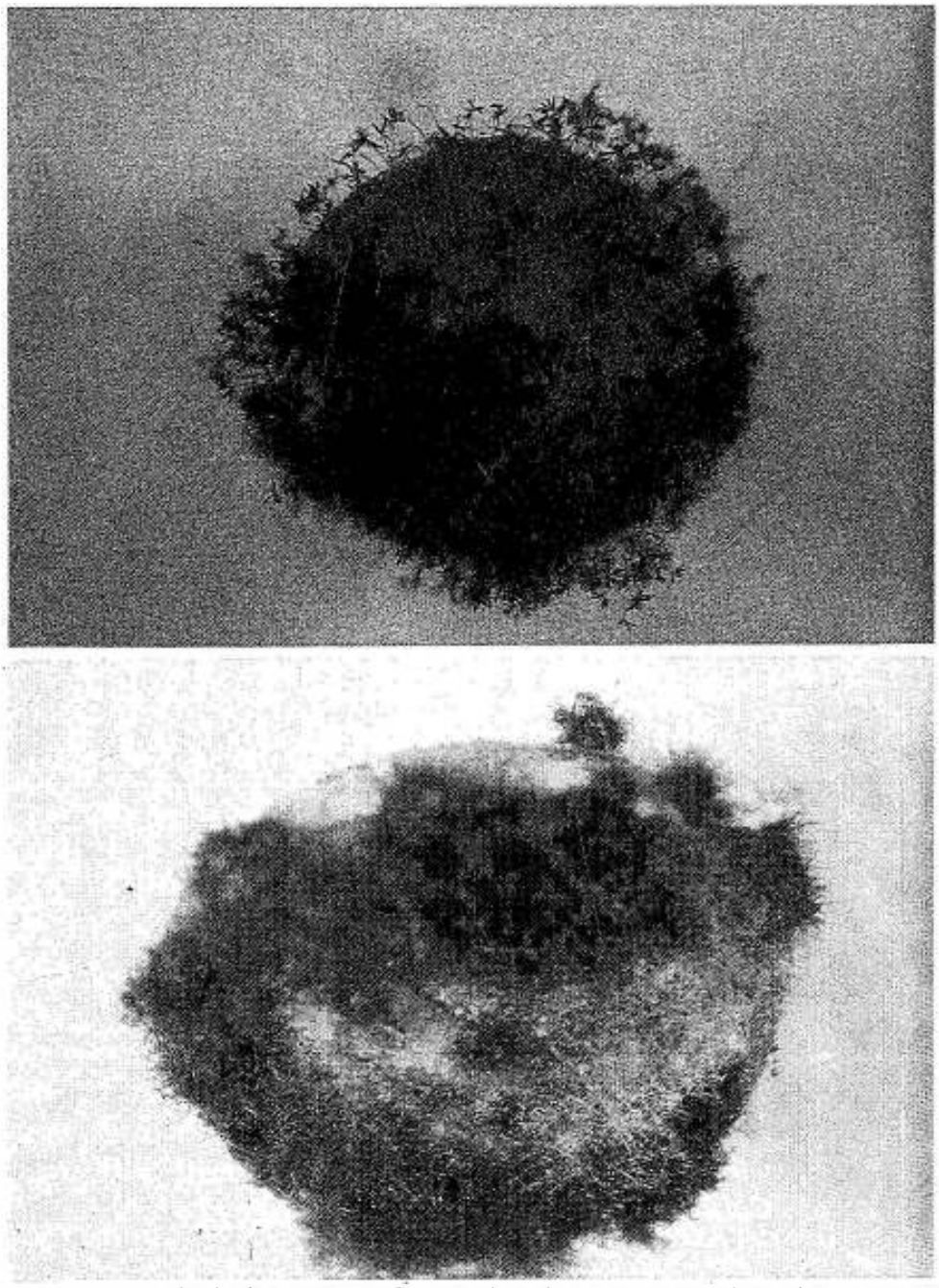


Figure 16B.
(b) hairy, velvety B. setariae *fungus on incubated finger millet seed, ×35,*
*(c) perithecial state (*Cochliobolus setariae*) of the fungus on pearl millet, ×113.*

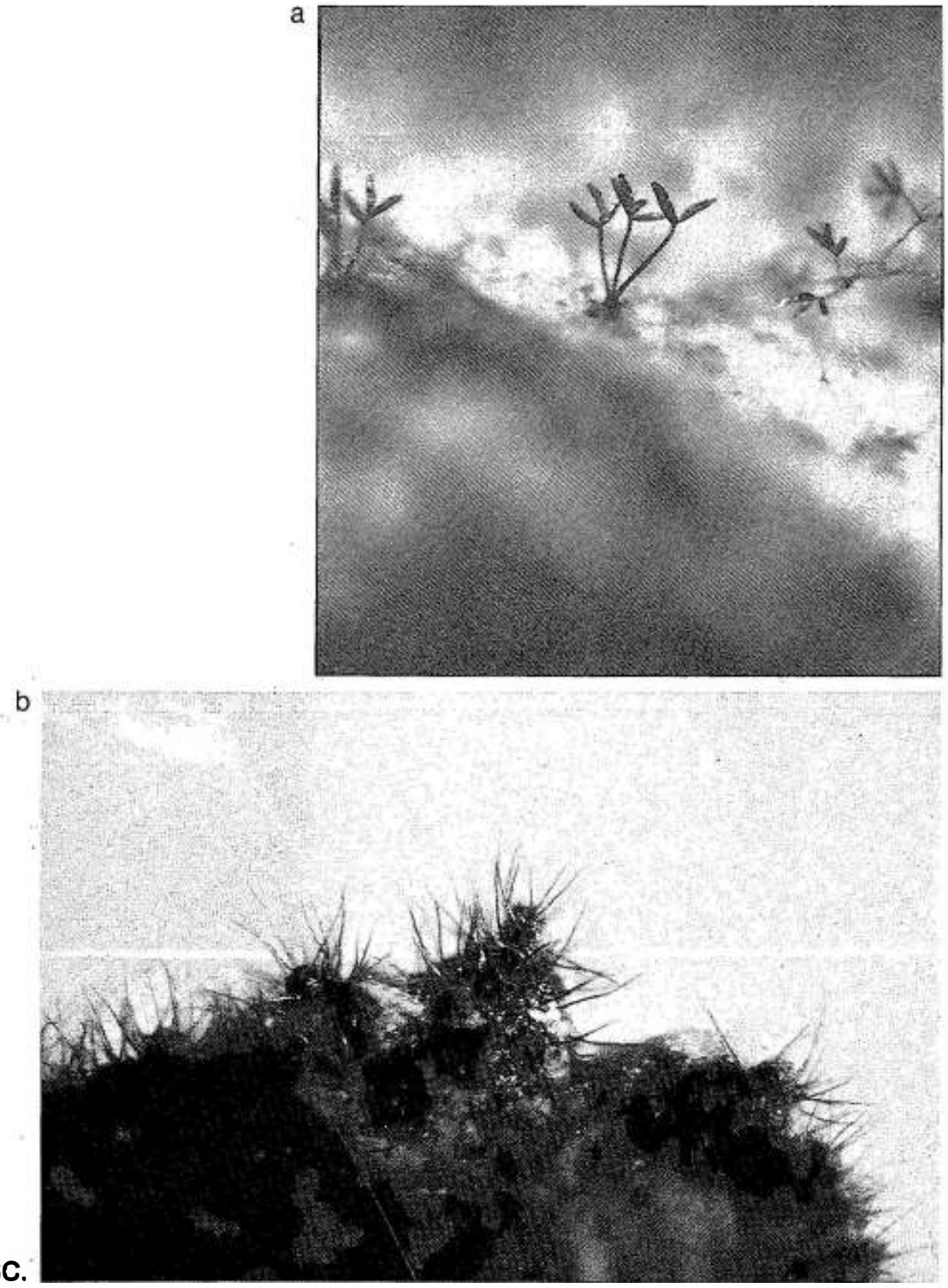


Figure 16C. *(a) Conidiophores, and conidia of* B. setariae, *×113, (b) perithecia of* C. setariae, *×98.*

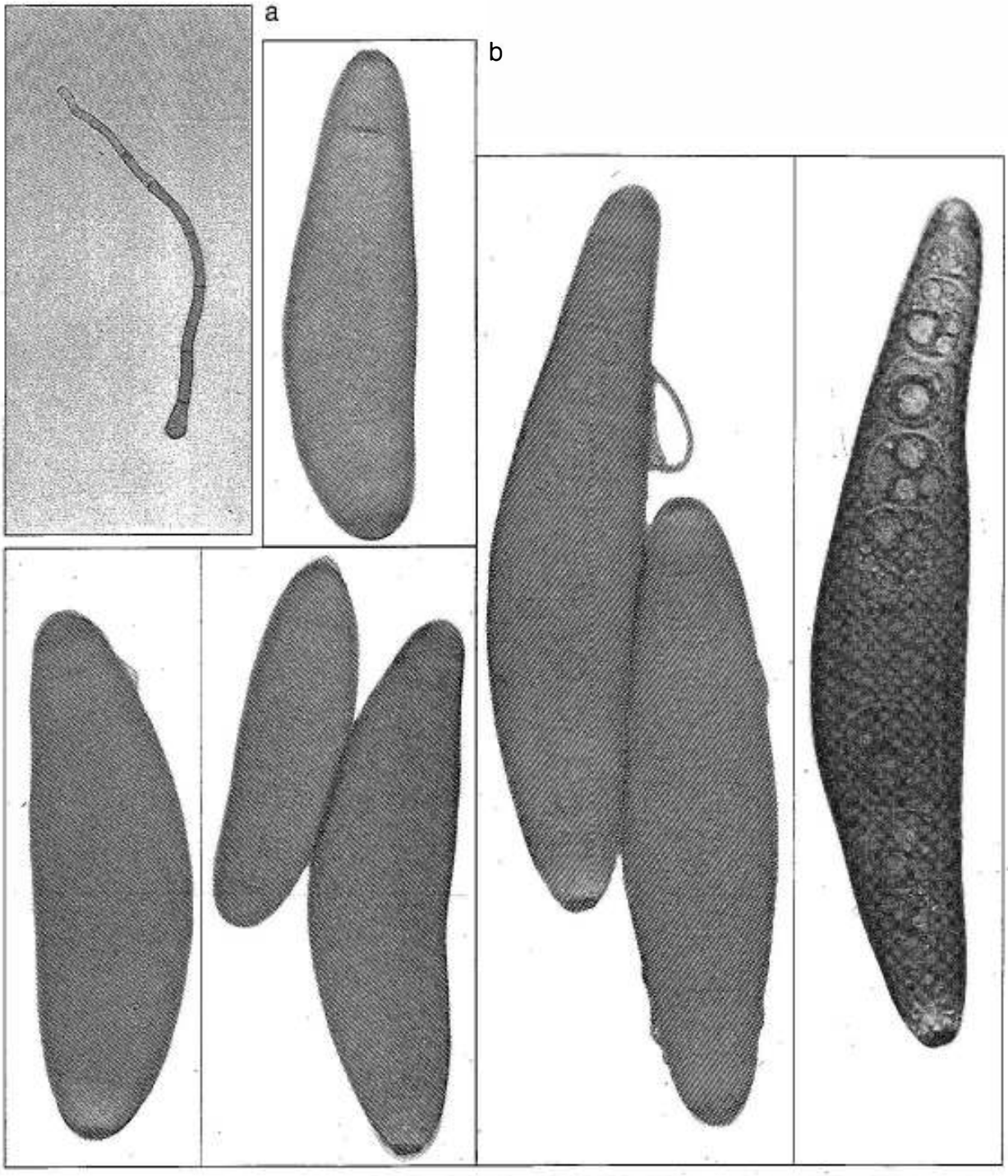


Figure 16D.
(a) Conidiophore, ×1130, (b) conidia, ×1130, of B. setariae (C. setariae),

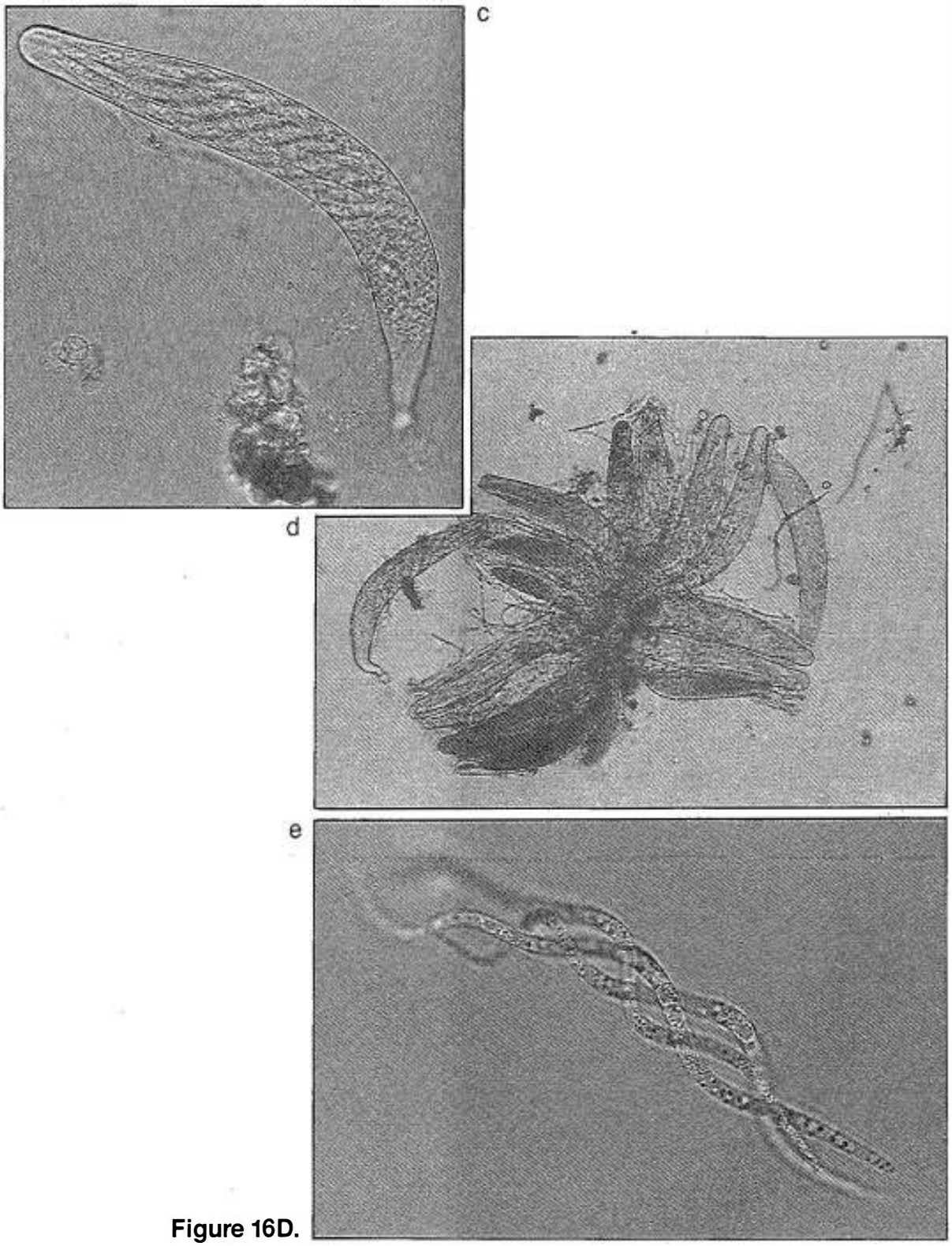


Figure 16D.
(c) ascus, ×451, (d) asci, ×251, (e) ascospores, ×451, of C. setariae (B. setaria).

Claviceps fusiformis Loveless

Claviceps microcephala (Wallr.) Tul.

Claviceps fusiformis causes ergot of pearl millet. In infected plants, the fungus produces sclerotia in place of normal grains. Admixtures of sclerotia, sown along with the normal seed, may serve as a primary source of disease inoculum. **Sclerotia** can be detected by visual examination. They are hard to brittle, weigh less than the normal seed, are light to dark brown, grayish-brown, or blackish-brown, pyriform, obpyriform, elongated to rounded, somewhat longer than the spikelets, with irregular longitudinal furrows, and are 3–6 mm long and 2–3 mm wide. The apical portion of the sclerotium tapers and is somewhat curved, while the basal portion is broad (Fig.17) (Butler 1918; Thakur 1984; Thakur et al. 1984).

Control. Avoid collecting seeds from endemic fields during pre-export crop health inspections. Whenever sclerotia are detected in the seed lot, separate them manually or by immersing the seed in 10% common salt solution. The sclerotia float on the surface of the salt solution and can be separated out (Nene and Singh 1976). Seeds that do not float should be dried and then given fungicidal treatment recommended for the export certification of pearl millet (see Appendix 3).

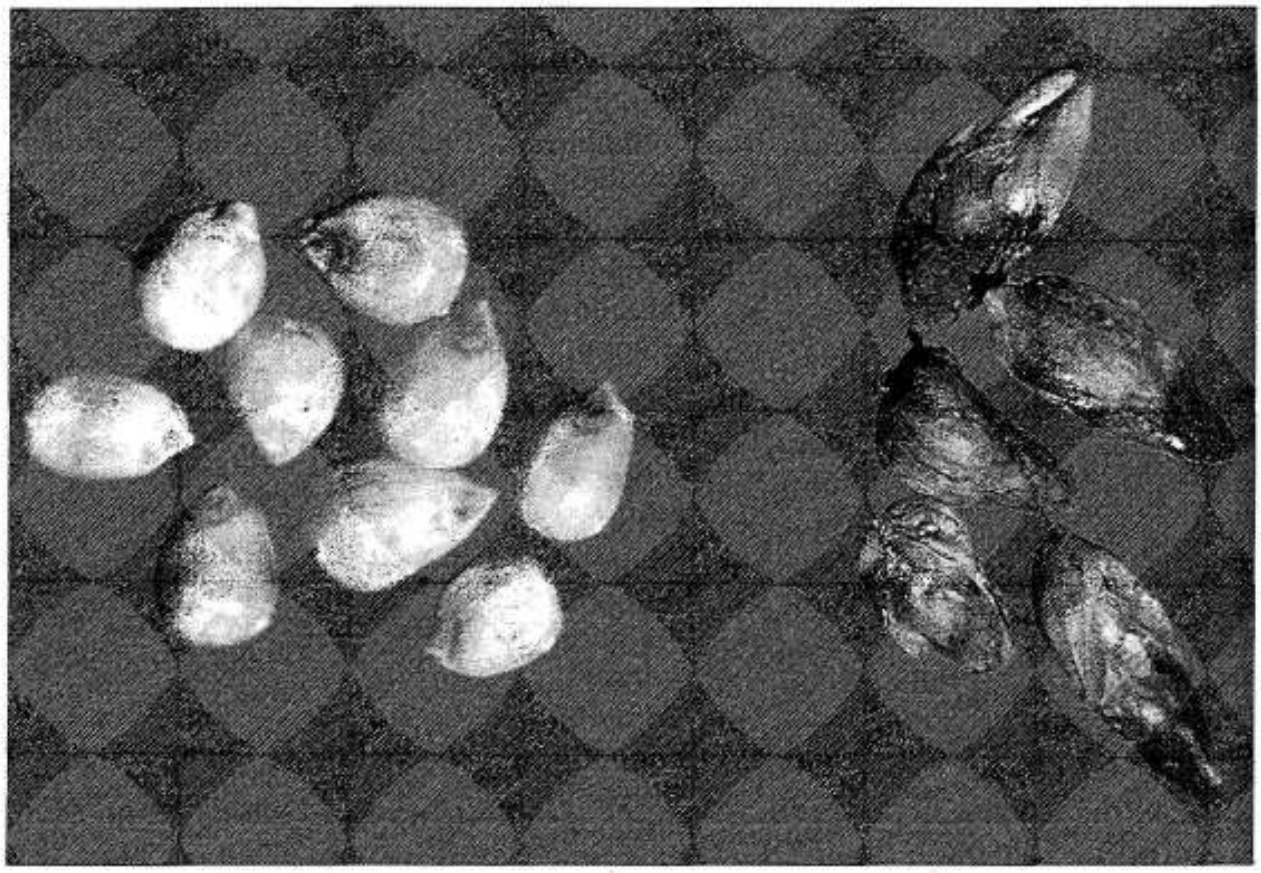


Figure 17.
Sclerotia of Claviceps fusiformis *(right) and healthy pearl millet seed (left),* ×*16.*

Curvularia penniseti (M. Mitra) Boedijn

This species of *Curvularia* causes leaf spots in sorghum and pearl millet. It may be present as dormant mycelium in infected seeds and can be detected by incubation tests. The fungus produces dark brown colonies on incubated seed (Fig.18A). The **mycelium** is mostly immersed in the seed coat and is composed of branched, septate, pale brown, thick hyphae. The hyphae are swollen at the point of origin of the conidiophore. **Conidiophores** arise singly or in groups and are erect, simple, straight or flexuous, sometimes geniculate, mid to dark reddish-brown and paler towards the apex, 68.4–200 μm long, 6–8 μm wide with a swollen base of 10–16 μm, and 3–5 μm wide at the apex (Fig.18B). **Conidia** are produced acropleurogenously. The 3-septate conidia are clavate and almost always slightly curved; septa are thick and visible at higher magnifications under the stereobinocular microscope. The third cell from the base of the conidium is larger than the others and is darker in color. Cells at either ends are subhyaline or pale and measure 29–42 × 13–20 μm (Fig.18C) (Mitra 1921; Ellis 1966; Sivanesan 1987).

Control. Discard moldy seeds during visual examination. Sorghum seed can be treated with thiram at about 2.5 g kg^{-1} (Agarwal and Khare 1978). For pearl millet seed, benzothiazole (Delsan-30® at about 1 mL kg^{-1}) is effective in eradicating the seedborne inoculum (Rao et al. 1984).

Figure 18A.
Infected pearl millet seed after incubation, showing the growth of Curvularia penniseti, *×36.*

Figure 18B.
Conidiophores and conidia of C. penniseti *growing on pearl millet seed, ×122.*

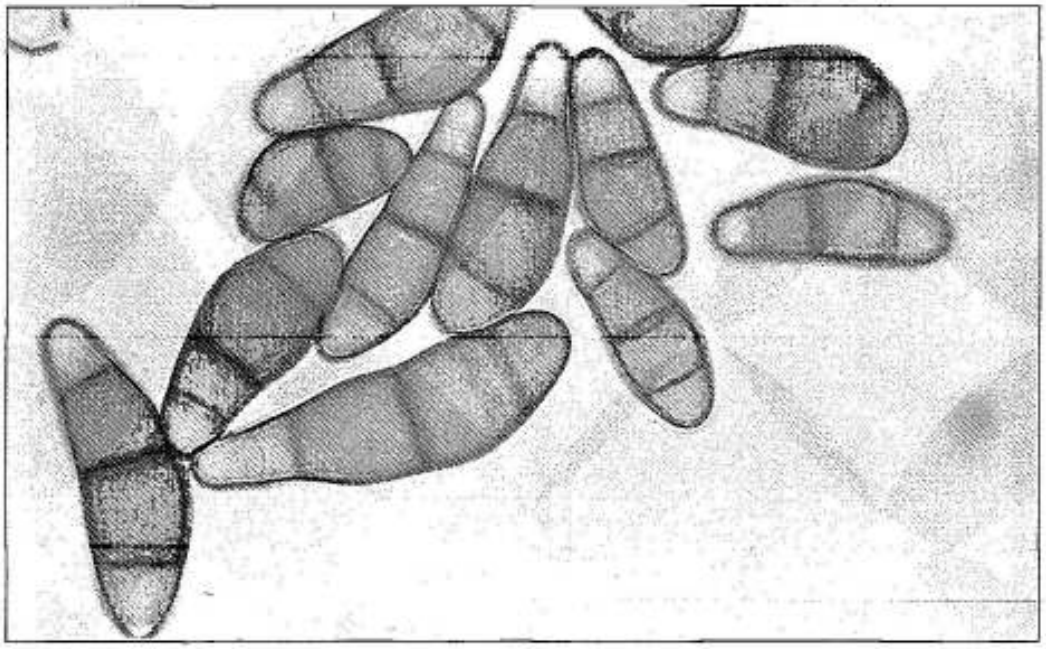


Figure 18C.
Conidia produced by C. penniseti, *×1130.*

Pyricularia penniseti Prasada and Goyal

Pyricularia penniseti causes brown leaf spot of pearl millet, and is known to survive in infected seeds. Seedborne infection can be detected by incubation tests. The fungus grows on incubated seeds, producing small, thin, hairy, inconspicuous gray-green to grayish brown, or olivaceous brown colonies (Fig.19A). The **mycelium** is sparse and immersed in the seed coat. **Conidiophores** are visible under a stereoscopic microscope growing directly on the seed coat often on the ends of the seed in small groups (Fig.19B). They are slender, thin-walled, simple or rarely branched, septate with a somewhat swollen basal cell and geniculated apex. Conidiophores bear conidia at their tips and successive growing points in a scorpoid manner, and measure 49.5–150.0 × 3.6–5.5 μm (Fig.19Ca). **Conidia** are hyaline, tri-celled, pyriform, narrow towards the apex, straight or slightly bent, obtuse to subobtuse at the tip, 18.35–36.70 × 7.35–11.10 μm with a small and distinct hilum at the base. The protuberant hilum indicates their point of attachment to the conidiophore. The walls of the conidia are smooth, 1–2 septate, and their cells are broadest at the lower septum (Fig.19Cb). Strong stereobinocular microscope lamps used very close to the colonized seed can dry delicate *Pyricularia* colonies, so if infection by this fungus is suspected the use of such lamps should be avoided. A minimum magnification of ×40 is recommended for identification (Prasada and Goyal 1970; Singh and Pavgi 1977).

Control. Seed treatment with captan or quintozene (Brassicol®) at about 1.5 g kg^{-1} is advised to reduce the seedborne inoculum (Duhan and Thakur 1977).

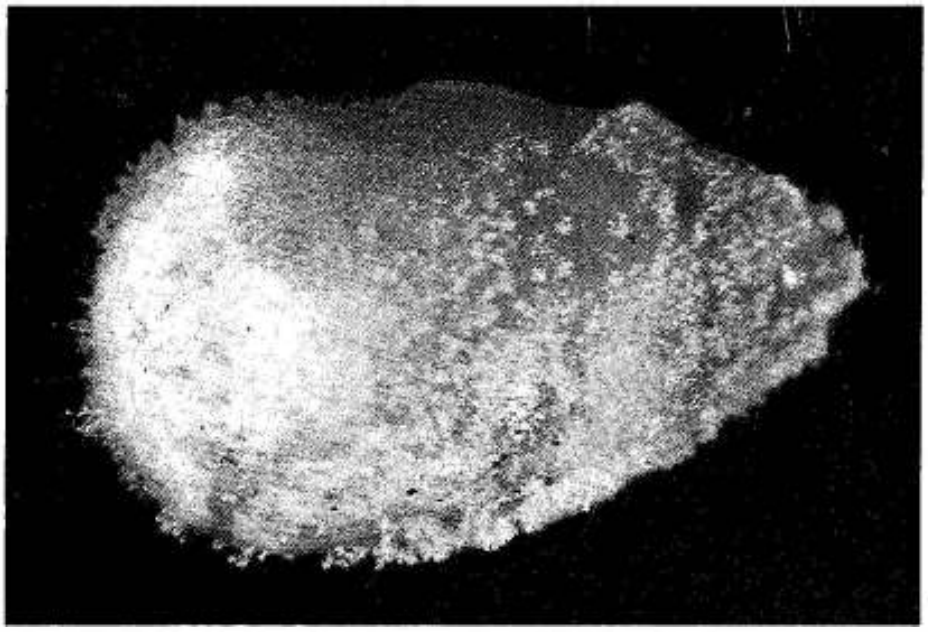


Figure 19A. *Infected pearl millet seed after incubation, showing the growth of* Pyricularia penniseti, *×46.*

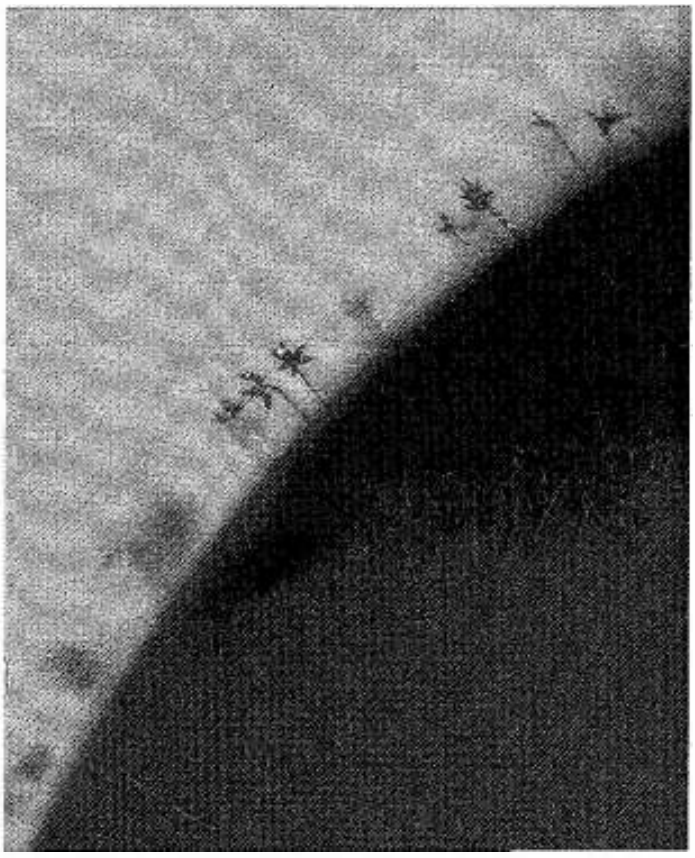


Figure 19B. *Conidiophores and conidia of* P. penniseti, *×57.*

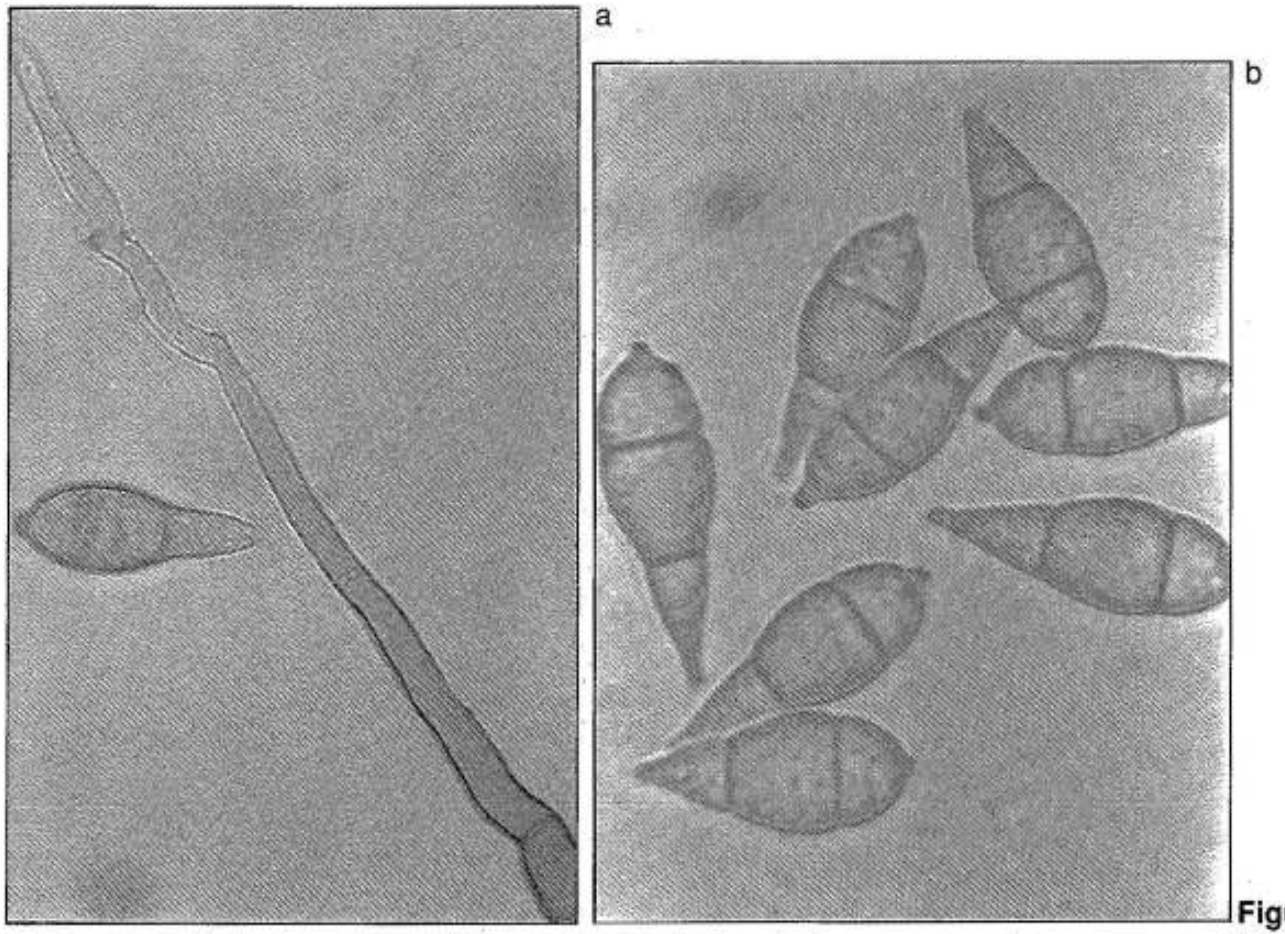


Figure 19C.
(a) Conidiophore, ×1130, and (b) conidia of P. penniseti, *×1130.*

Sclerospora graminicola (Sacc.) J. Schroet.

This fungus is an obligate parasite and causes downy mildew of pearl millet. It is reported as internally and externally seedborne; however, some workers do not agree that it is internally seedborne. Seeds harvested from infected fields are likely to carry the fungus. The fungus is seed-transmitted as oospores adhering to the seed coat and as dormant mycelium present in internal seed tissues. During seed health inspection, oospores can be detected by a seed-washing test. Under a compound microscope, **oospores** (Fig. 20) appear light to reddish-brown, and are globose, smooth, thick-walled, mostly uniformly thick, and 20–40 µm in diameter. Mature oospores are spherical and have three walls; the exosporium, which is smooth and is 2 µm thick, mesosporium, and endosporium. The oogonial wall is persistent and is visible as irregular folds on the mature oospore (Butler 1907; Kenneth 1975; Safeeulla 1976; Shetty et al. 1978 and 1980; Francis and Williams 1983a).

Control. Conduct pre-export crop health inspections during the growing season and select seed from disease-free fields. The seed treatment schedule recommended for the export certification of pearl millet is given in Appendix 3, effectively eradicates the seedborne inoculum (India 1977; Williams and Singh 1981).

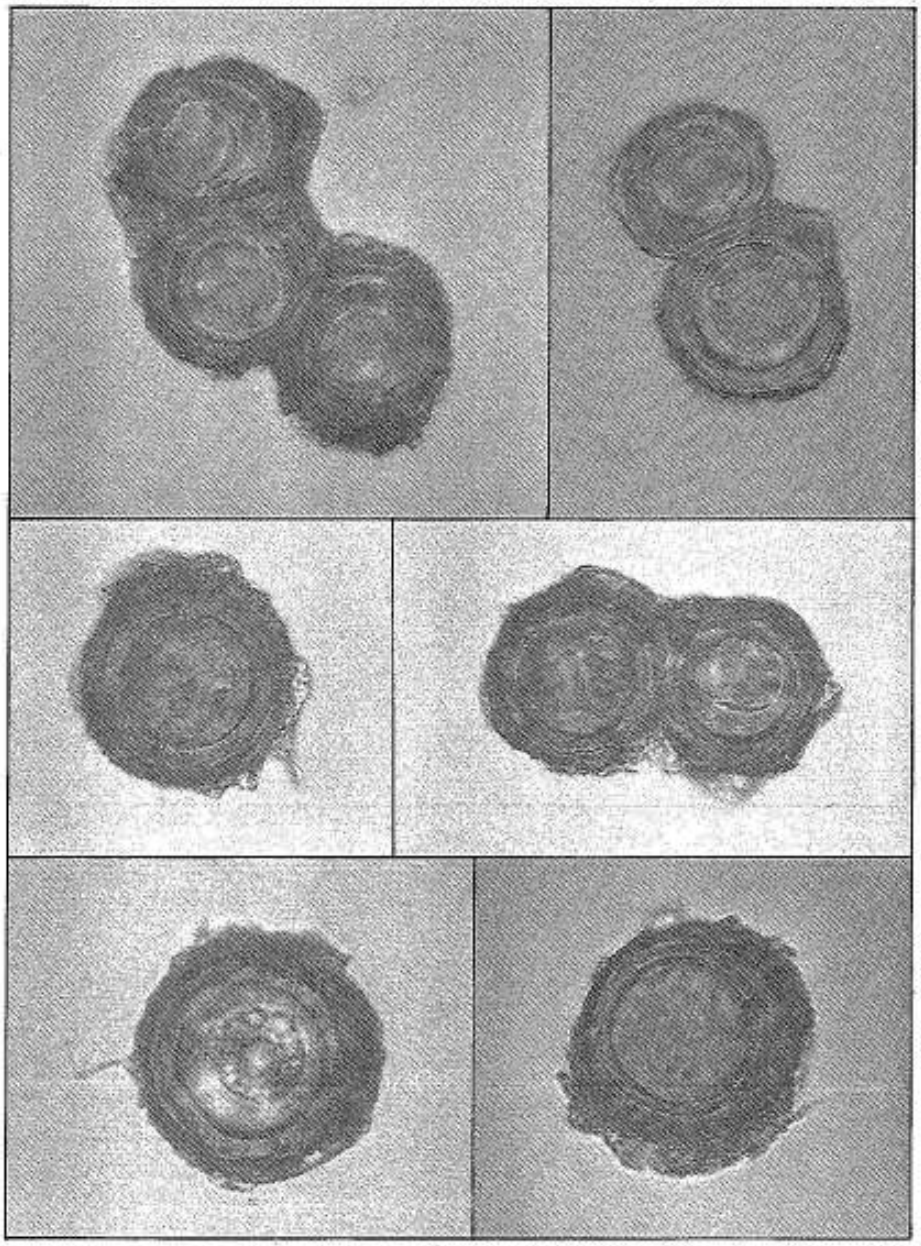


Figure 20.
Oospores of Sclerospora graminicola, *×451.*

Tolyposporium penicillariae Bref.

Tolyposporium penicillariae causes smut of pearl millet. Smut **sori** are usually found in pearl millet seed originating from smut-infected fields. These sori are produced in the ovaries of panicles infected by *T. penicillariae*. In addition to the admixtures of smut sori, healthy-looking seed may also become contaminated by aerially disseminated spores, when smut sori rupture in the field during threshing. Teliospores, when present on the surface of the seed, can be detected by a seed-washing test. Smut **sori** are visible to the naked eye, and can be readily seen under a magnifying lens (Fig.21A). They are pear-shaped, 3–5 mm long, and 2–3 mm broad, i.e., half to twice the diameter of normal pearl millet grains. The apex of the sorus is bluntly round to conical. The sori are chocolate brown to dark brown, often dirty black in color, especially when mature. **Teliospores** occur in large numbers and are attached to one another, forming compact balls of varying shapes, 50–150 µm in diameter. Individual teliospores are globose to subglobose, or ovoid, light brown, with smooth, irregular or slightly roughened walls, single celled, and 6–12 µm in diameter (Fig.21B) (Butler 1918; Bhatt 1946; Ainsworth 1965e).

Control. Methods recommended for control of long smut of sorghum (*T. ehrenbergii*) can be used during phytosanitary inspections. Seed treatment is useful to prevent the introduction of smut spores from contaminated seed to clean soil. Seed treatment with ceresan is effective to eradicate the externally adhering spores on contaminated seeds (Rachie and Majumdar 1980).

Figure 21A.
Smut sori produced by Tolyposporium penicillariae *in pearl millet, ×11.*

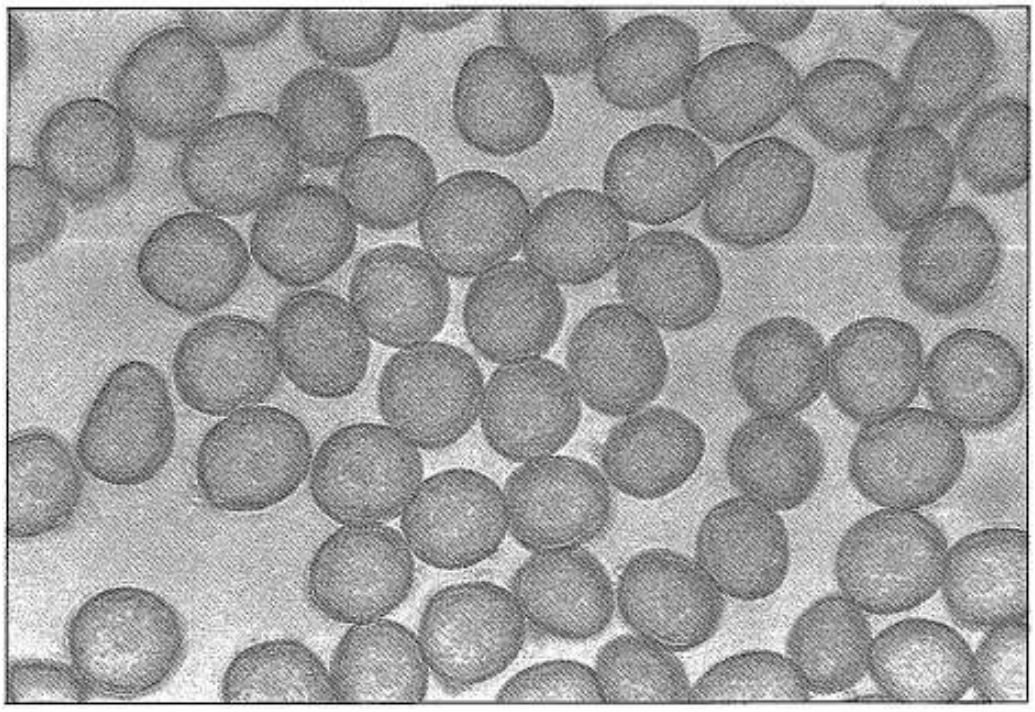


Figure 21B.
Teliospores of T. penicillariae, *×1130.*

Finger Millet

Bipolaris nodulosa (Berk. & M.A. Curtis) Shoemaker

Helminthosporium nodulosum Berk. & M.A. Curtis
Helminthosporium leucostylum Drechsler
Cochliobolus nodulosus Luttrell

Bipolaris nodulosa causes blight of finger millet. The fungus is a virulent parasite that attacks all parts of the plant, including the roots. Infected seeds can be identified by visual examination and incubation tests. Heavily infected dry seeds appear shriveled, are smaller, darker brown, and weigh less than healthy seeds (Fig.22A). The palae of infected seeds are gray and remain closed, whereas the palae of healthy seeds are yellow and open wide. Infected seed, when stored in humid weather, appears sooty or olivaceous green, due to the superficial growth of the fungus. Infected seeds usually fail to germinate, and even if they do, produce decayed seedlings. Thick, dark gray hyphae can be seen on the glumes, palae, and seed coats of less severely infected seeds. On incubated seed, the **mycelium** consists of thin-walled septate hyphae, which are hyaline, even at the place where conidiophores are produced (Fig.22B). **Conidiophores** occur singly or in groups. They are subhyaline, light olivaceous, geniculate, hyaline at either end, unbranched, or sometimes produce short lateral branches, usually 3–8 septate, have swollen subhyaline basal cells, with flat or anvil-shaped tips, and measure 21–112 × 4–6 μm (Fig.22C). Conidia are borne at a distance of 18–40 μm from the base, and successive spores at intervals of 5.25 μm. **Conidia** are olive green, light olive brown, straight, obclavate, ovoid, obovoid, widest at the second septum, their apices taper uniformly to form narrow tips which are rounded off abruptly. The hilum is dark, conspicuous but not protruding, 1–8 septate, and 18–80 × 10–21 μm (Fig.22D) (Mitra and Mehta 1934; Ramakrishnan 1963; Ellis and Holliday 1972; Sivanesan 1987).

Control. Conduct pre-export crop health inspection and avoid collecting seed from endemic areas. Seed treatment to reduce the seedborne inoculum is effective with such fungicides as; dithianon (Delan WP75®) at about 2 g kg^{-1} (Rajashekar et al. 1989) in the form of slurry, guazatine (Panoctine-R® at about 2 mL kg^{-1}) (Ranganathiah and Rao 1982), or captafol at about 2 g kg^{-1} (Krishna Prasad and Basuchaudhary 1987).

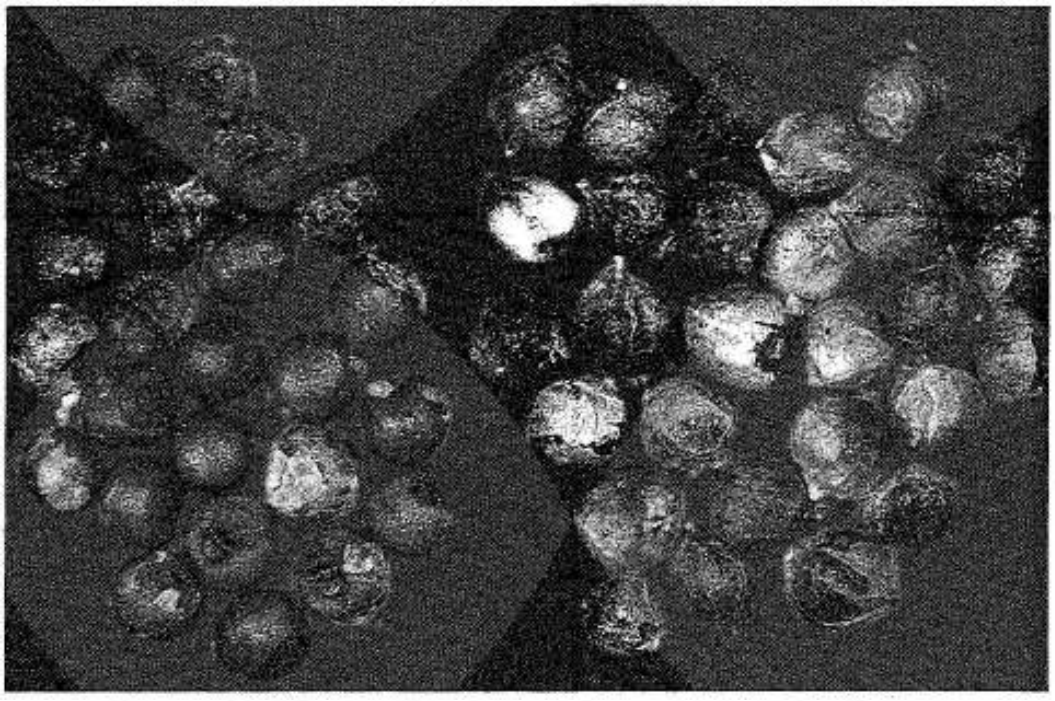


Figure 22A.
Dry seeds of finger millet showing damage caused by Bipolaris nodulosa *(right) and healthy seeds (left), ×5.*

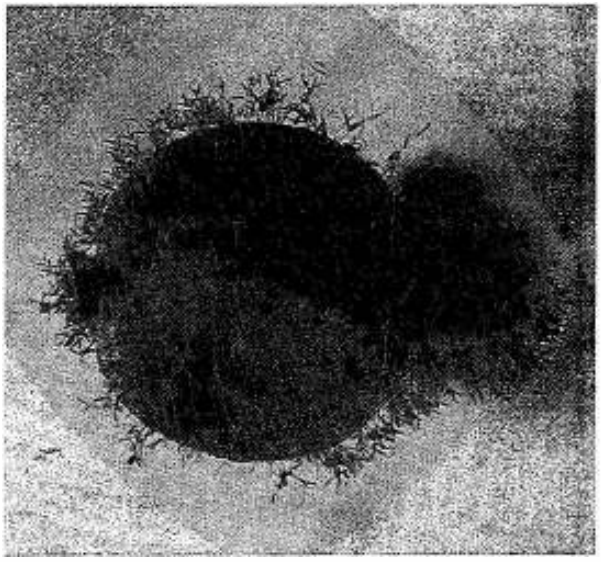


Figure 22B.
Infected seed after incubation, showing the growth of B. nodulosa, *×20.*

Figure 22C.
Conidiophores and conidia produced by B. nodulosa, *×113.*

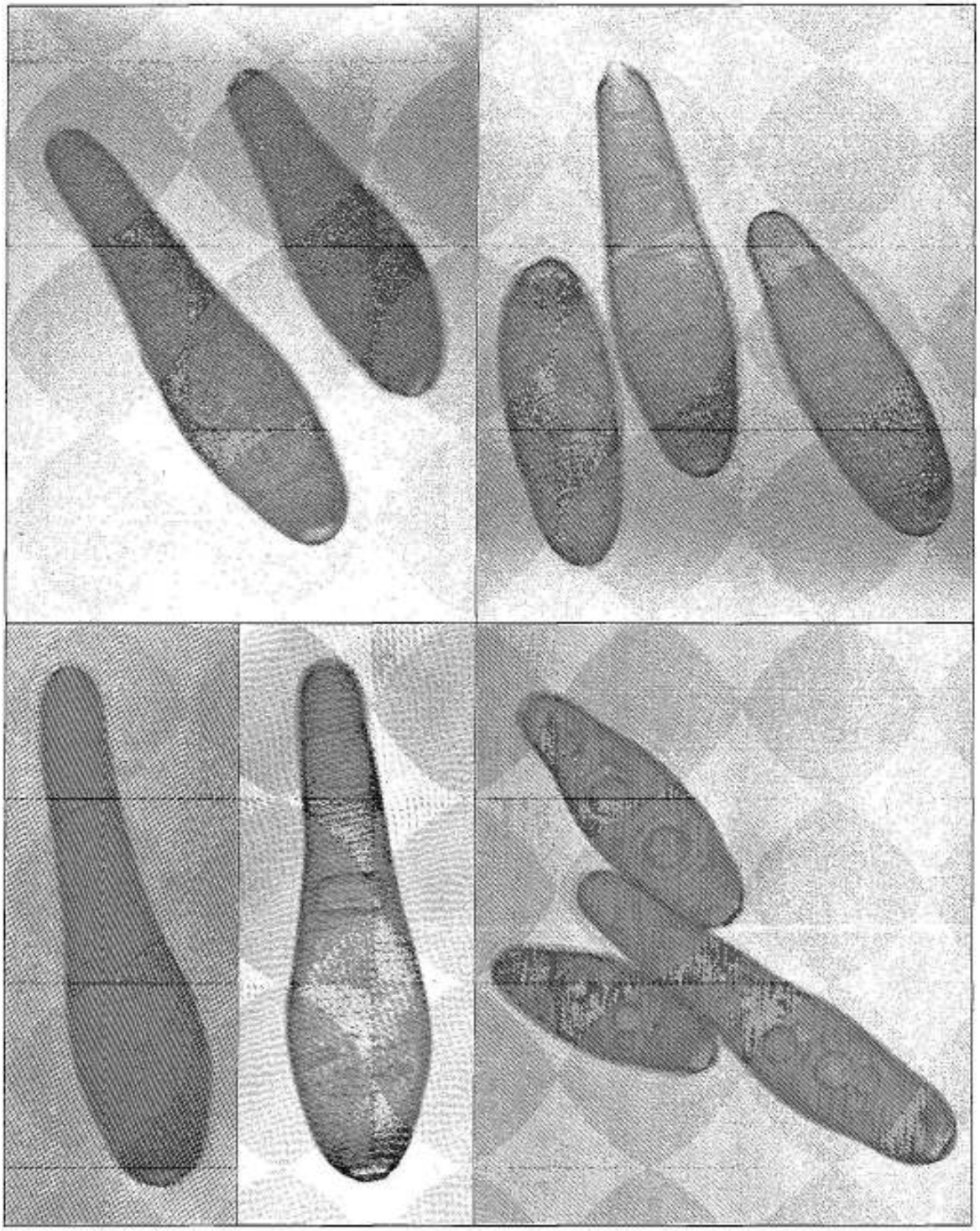


Figure 22D.
Conidia produced by B. nodulosa*, with a hyaline zone just above the hilum, ×1130.*

Melanopsichium eleusinis (Kulk.) Mundkur & Thirum.

Ustilago eleusinis Kulkarni

Melanopsichium eleusinis causes smut of finger millet. This fungus is ovaricolous, and the infected grains are scattered in the spike. During seed distribution, the fungus can spread in the form of smut sori, or as teliospores adhering to the surface of the seed. Smut sori are visible to the naked eye. They are round, elongated or irregularly shaped bodies projecting beyond the glumes. Smut sori are from one to six times the diameter of normal seed. Usually they are 3–8 mm in diameter when round, and 4–15 mm long when elongated (Fig. 23A). **Sori** are chocolate brown or dirty black. Their color is due to the membrane enclosing the spore mass which is deep brown to black. Sori contain lysigenous cavities in the hypertrophied host tissue which can be seen under a compound microscope. The cavities are irregular in shape and size with no columella, and contain powdery black spore masses. **Teliospores** adhering to healthy-looking seed can be detected by a seed-washing test. They are globose to subglobose with a densely echinulated epispore. The spores are olivaceous brown, rough surfaced, and 6–12 μm in diameter (Fig. 23B). The outer layer of the exosporium is highly pigmented and bears projections (Mundkur and Thirumalachar 1952; Khanna and Payak 1971).

Control. Conduct pre-export crop health inspections and avoid collecting seeds from endemic fields. Discard smut sori during visual examination. Seed dressing is useful to prevent the introduction of smut spores from contaminated seed to clean soil. Soaking seeds in 2% copper sulphate solution kills smut spores on the seed surface (Kulkarni 1922).

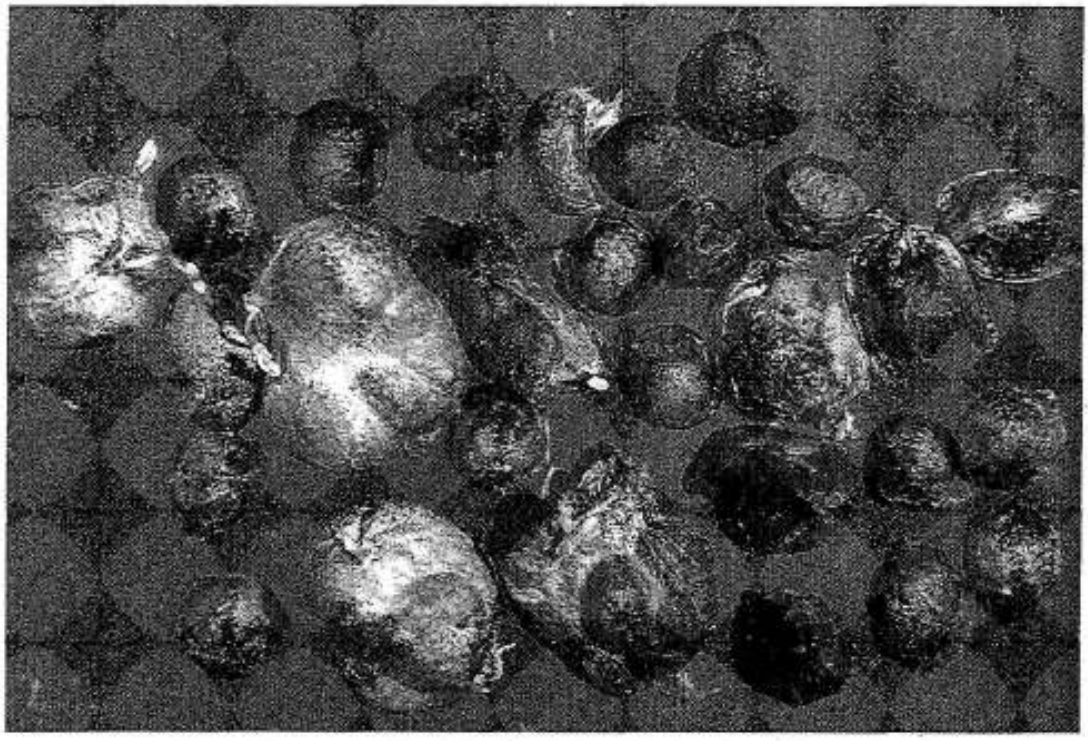


Figure 23A.
Smut sori produced by Melanopsichium eleusinis, *and the contaminated seeds of finger millet,* ×*4.*

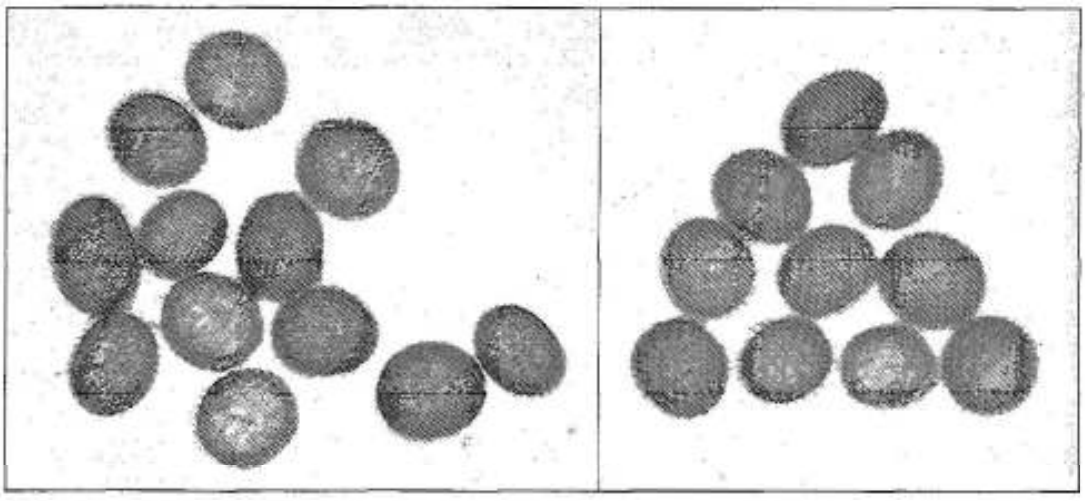


Figure 23B.
Teliospores of M. eleusinis, ×*1130.*

Pyricularia setariae (Wallace) Ramakrishnan

Pyricularia grisea (Cooke) Sacc.

Pyricularia setariae causes leaf and neck blast of finger millet. *Pyricularia* can infect the crop at all stages from seedling to grain formation. Infected seeds can be detected by visual examination and incubation tests. Dry infected seeds of finger millet appear brown or dark gray, shriveled, undersized, and a high percentage of them fail to germinate. Upon incubation, infected seeds show profuse growth (Fig. 24A) of mycelium and spores. Colonies are colorless to light olive green, and grow in tufts or a continuous mass. The **mycelium** is profuse and darkens as it matures. Individual hyphae are hyaline, brown with hyphal cells somewhat swollen and 1.5–6.0 µm long. **Conidiophores** are simple, septate, darker at the base and paler towards the apex (Figs. 24B and 24Ca). **Conidia** are typically obpyriform, subhyaline, acrogenous, or produced one after another by the sympodial growth of the conidiophore. Each spore is 3–4 celled, the middle cell being broader and darker than the others, and measures 19–31 × 10–15 µm (Fig. 24Cb). The darker color of the middle cell is only observed in a few conidia. **Chlamydospores** are thick-walled, olive brown or dark brown, terminal or intercalary, and 4–10 µm in diameter (Wallace 1950; Ramakrishnan 1963).

Control. Conduct pre-export crop health inspections and avoid collecting seed from endemic areas. Seed treatment with organomercurial fungicides at about 2 g kg^{-1} is likely to reduce the seedborne inoculum (Ramakrishnan 1963). Guazatine (Panoctine-R® at about 2 mL kg^{-1} of seed) is also effective to reduce the seedborne inoculum (Ranganathiah and Rao 1982).

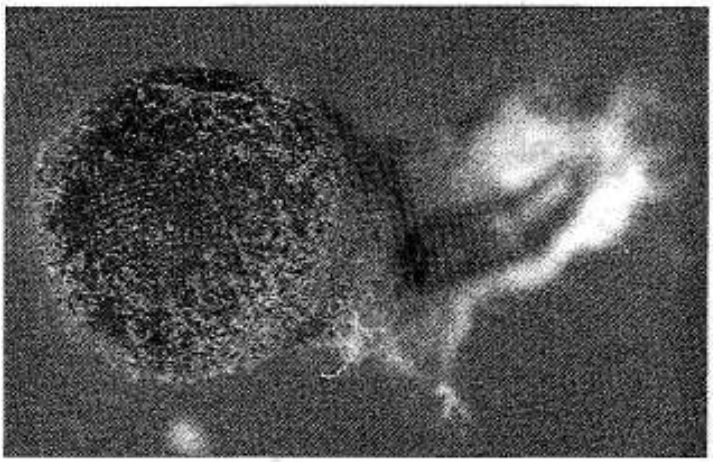


Figure 24A.
Infected seed of finger millet showing profuse growth of Pyricularia setariae, *×112.*

Figure 24B.
Conidiophores and conidia of P. setariae, *×113.*

a

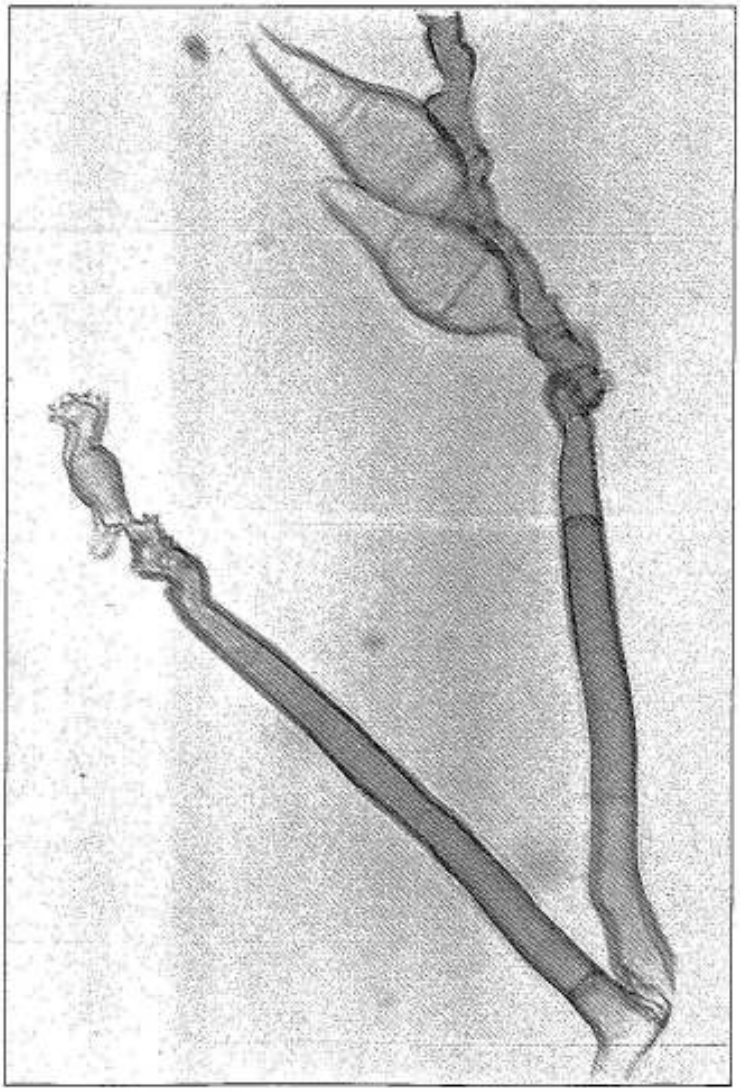


Figure 24C.
(a) Conidiophores, ×1130, and

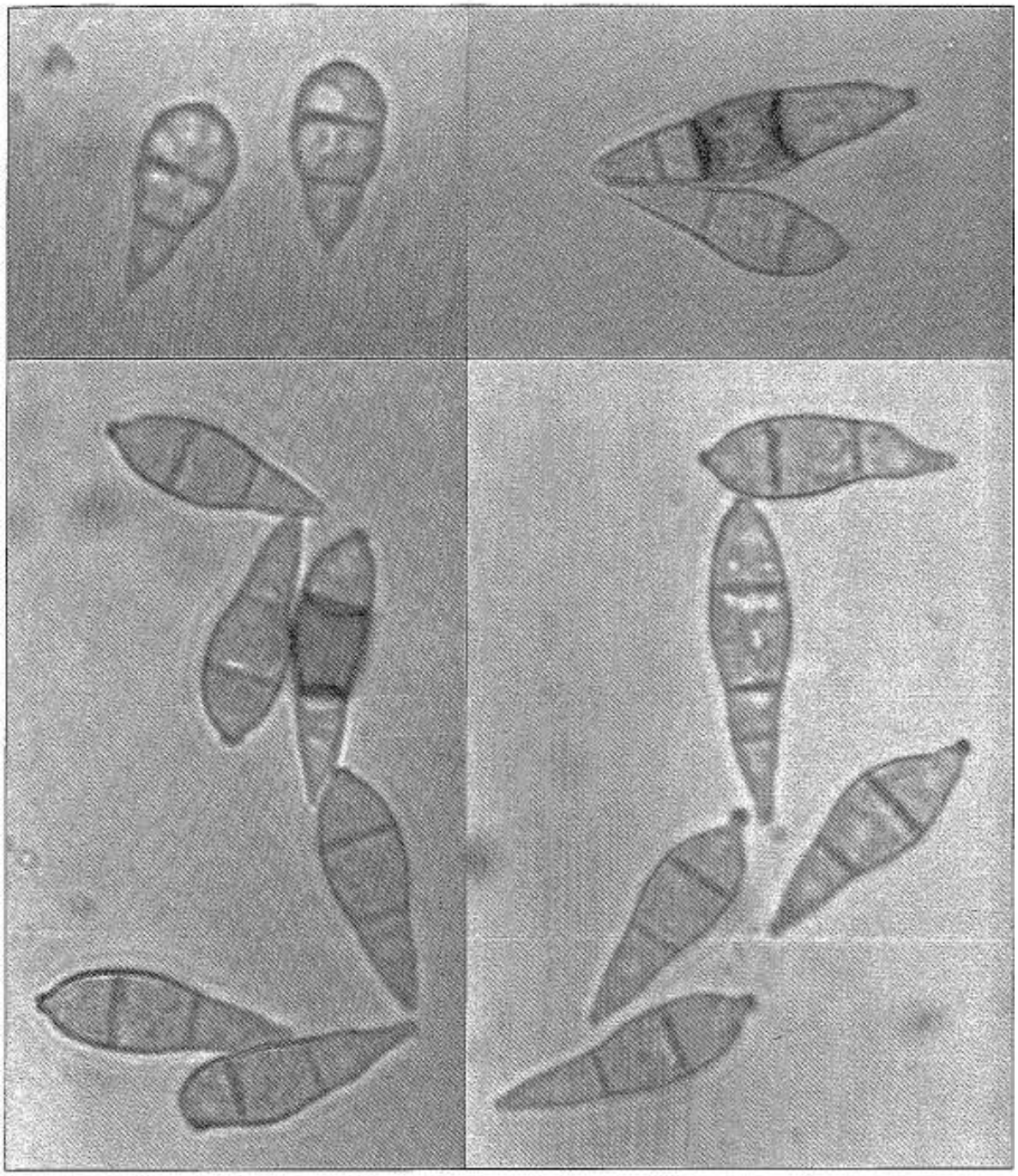


Figure 24C.
(b) conidia of P. setariae, *×1130.*

Sclerophthora macrospora (Sacc.) Thirum., Shaw and Naras.

Downy mildew of finger millet is caused by *Sclerophthora macrospora*. The fungus has more than 140 graminaceous hosts, but physiological specialization is well developed in this species, and as a result, certain races may be host-specific. The fungus may be associated with otherwise healthy seed in the form of oospores adhering to the surface, and dormant mycelium present in the embryonic tissue. **Oospores** adhering to the seed surface can be detected by a seed-washing test. They are spherical, dark to golden brown, thick-walled, the walls having three layers, with the outermost layer having small protrusions, and usually confluent with the host cell wall (Fig. 25). The oogonial wall is 2–3 μm thick and the oospores are 35–70 μm in diameter (Safeeulla 1976; Thirumalachar et al 1953).

Control. Finger millet seeds should be obtained from disease-free areas or crops inspected during the active growth phase (Ramakrishnan 1963; Rachie and Peters 1977). Information on the fungicidal seed treatment for eradication of the seedborne inoculum is not available.

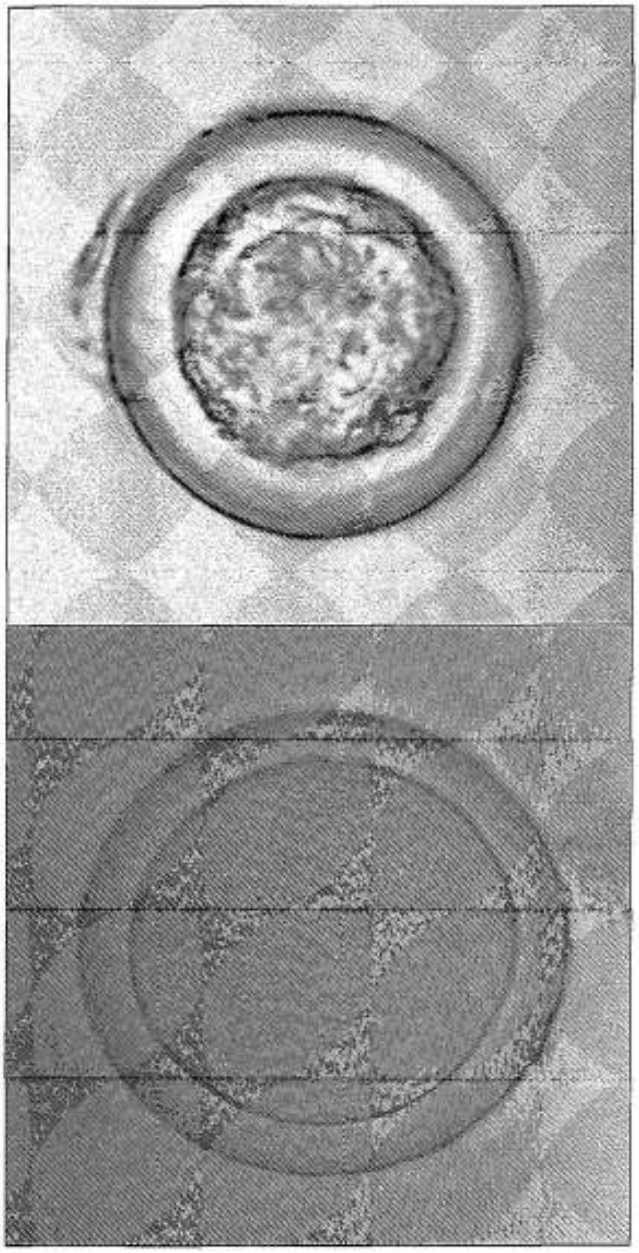


Figure 25.
Oospores of Sclerophthora macrospora, *×1130.*

Chickpea

Ascochyta pinodes L.K.Jones

Ascochyta pinodes is seedborne in chickpea, and causes blight. Infected seeds can be detected by incubation tests. The fungus produces numerous pycnidia on incubated seed; the pycnidia arising singly or in confluent masses resulting in reduced seed germination (Fig.26A). During blotting incubation tests **pycnidia** are often seen on the seed surface touching the blotting paper. They are solitary or in groups, immersed, erumpent or superficial, 100–200 μm in diameter, dark brown to black, globose, papillate, ostiolate, and conidia ooze out of them in light buff to flesh-colored gelatinous masses (Fig.26B: inset, at left). **Conidia** are hyaline, guttulate, smooth, ellipsoidal, slightly constricted at the septa, usually 1–septate, sometimes 2–3 septate, and measure 8–15 × 3–4.5 μm (Fig.26Ca). The perfect state of this fungus, *Mycosphaerella pinodes* (Berk. & Bloxam) Vestergr., also occurs on infected chickpea seeds; pseudothecia can be seen growing superficially or embedded in the seed coat. **Pseudothecia** arise singly, and are 90–180 μm in diameter, globose, dark brown to black with prominent beaks and apical papillate ostioles (Fig.26B: inset, at right). They contain numerous asci. The **asci** are cylindrical to subclavate, bitunicate, short, stipitate or sessile, hyaline, and are 50–80 × 10–15 μm (Fig.26Cb and inset). Each ascus contains eight **ascospores** which are hyaline, bicelled, constricted at the septum and rounded at the base, irregularly biseriate, ellipsoid, biguttulate, and measure 12–18 × 4–8 μm (Fig.26Cc) (Punithalingam and Holliday 1972a; Bretag and Mebalds 1987).

Control. Chickpea seeds intended for export should be collected from disease-free areas or fields inspected during the active growth phase. Information on specific seed-dressing fungicides to be used on chickpea is not available.

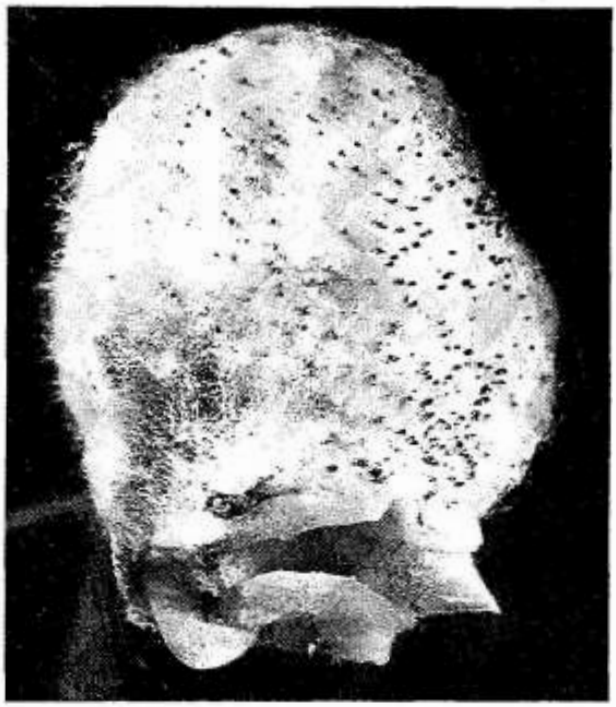


Figure 26A.
Infected chickpea seed after incubation, showing the growth of Ascochyta pinodes *and its perfect state* Mycosphaerella pinodes, *×13.*

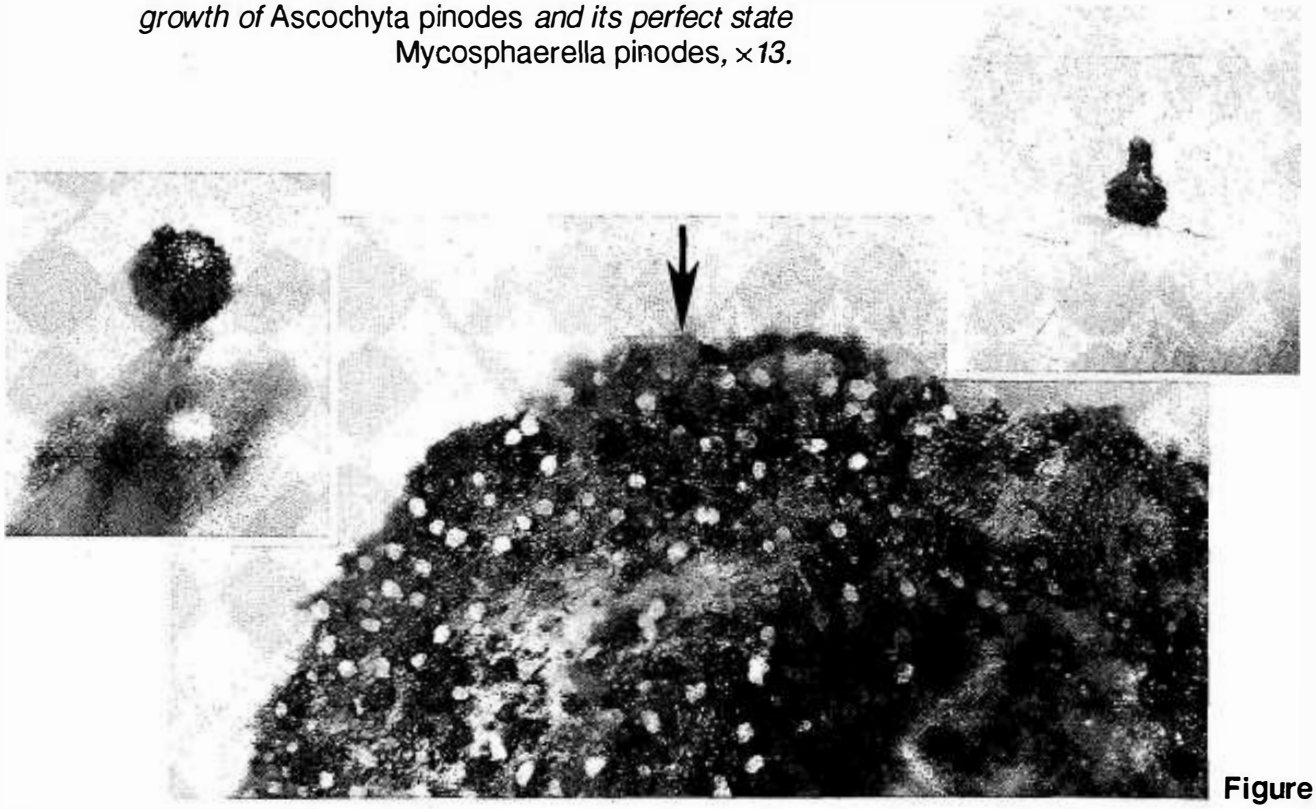


Figure 26B.
Infected chickpea seed after incubation, showing pycnidial (carrot red, arrowed) and pseudothecial (light yellow) oozes, ×36. Inset (left) pycnidium, ×88; inset (right) pseudothecium, of A. pinodes (M. pinodes), *×80.*

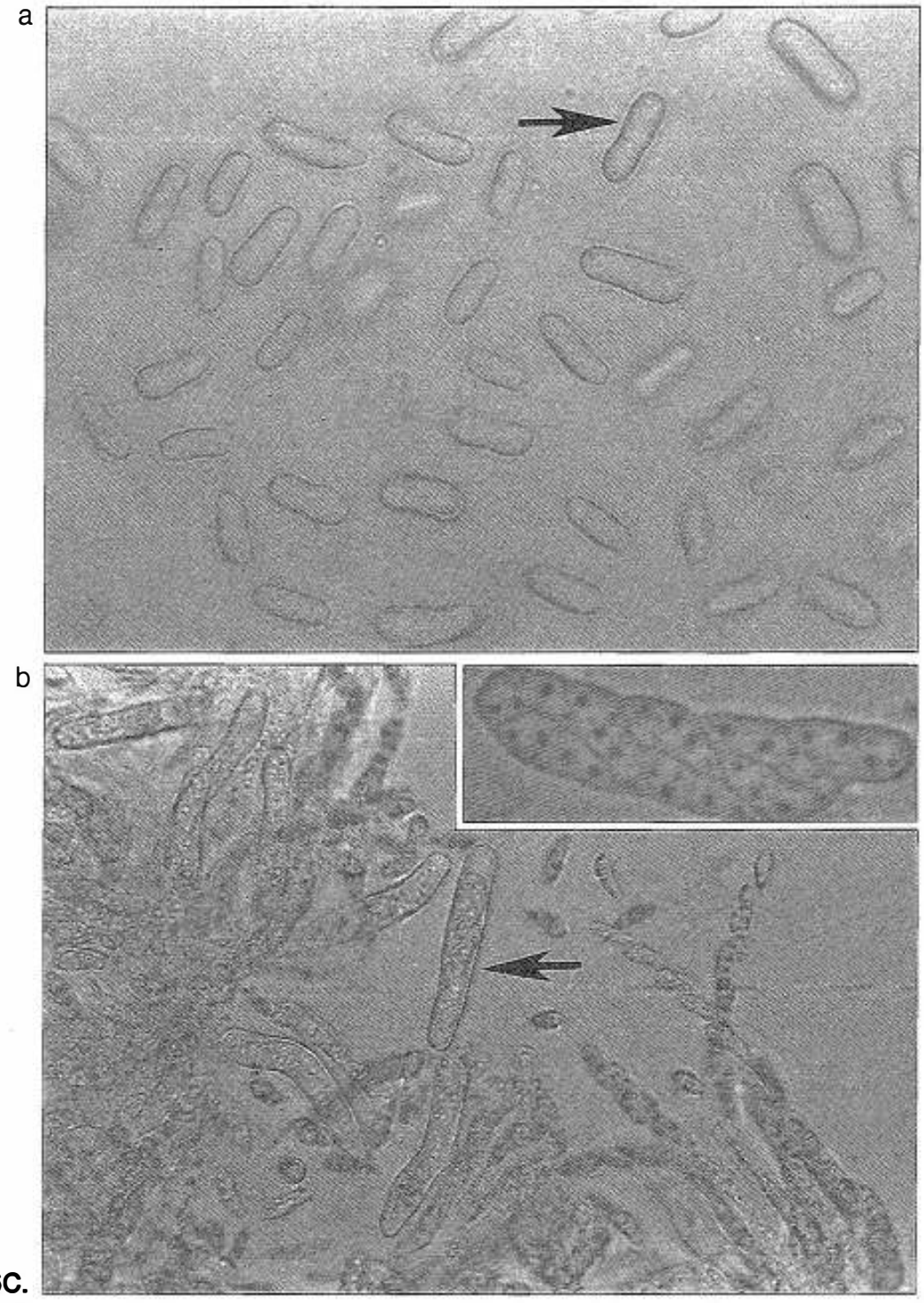


Figure 26C.
(a) Bicelled conidia (arrowed) of A. pinodes, *×1130, (b) asci (arrowed), ×451, inset, ascus, ×1130,*

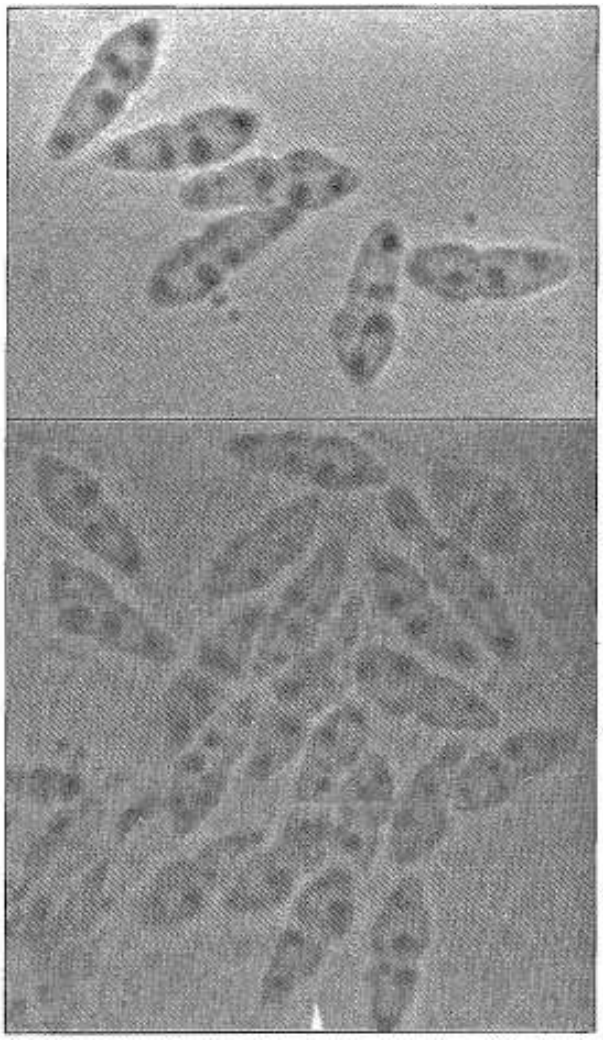


Figure 26C.
(c) ascospores of M. pinodes, *×1130.*

Ascochyta rabiei (Pass.) Labrousse

Phoma rabiei (Pass.) Khune & J.N.Kapoor

Ascochyta rabiei causes ascochyta blight of chickpea. It is an internally and externally seedborne pathogen. Infected seeds can be detected visually and by incubation tests. Infected dry seeds are small, wrinkled, weigh less than healthy seed, and often fail to germinate. Heavy fungal invasion results in dark lesions of various shapes and sizes on the seed. Concentric rings of pycnidia can be observed in the lesions. When the infection is deep, pycnidia rupture the seed coat (Fig.27A). The fungus grows slowly on incubated seed (Fig.27B) as mycelium and pycnidia that are usually formed in concentric rings and are seen particularly on the seed surface touching the blotting paper during the blotting incubation test. **Pycnidia** are spherical to globose or depressed, amphigenous, separate, confluent, dark brown, ostiolate, measuring 140–245 μm in diameter (Fig.27C). When the pycnidia mature, conidia ooze from them in a gelatinous cream-colored mass. **Conidia** are hyaline, 1–2 celled, biguttulate, straight or slightly curved, oval to oblong, rounded at each end, and 6–16 × 3.4–5.6 μm (Fig.27D). Most conidia are unicellular, and they may or may not be constricted at the septum (Kovachevsky 1936; Punithalingam and Holliday 1972b; Maden et al. 1975; Haware et al. 1986).

Control. Avoid collecting seeds from endemic areas and conduct pre-export crop health inspections during the growing season. Discard infected seeds during visual examination. Seed treatment with 11% tridemorph + 36% maneb (Calixin-M®) (Reddy et al. 1982), or thiabendazole at about 3 g kg^{-1} eradicates the seedborne inoculum (Reddy and Kababeh 1984).

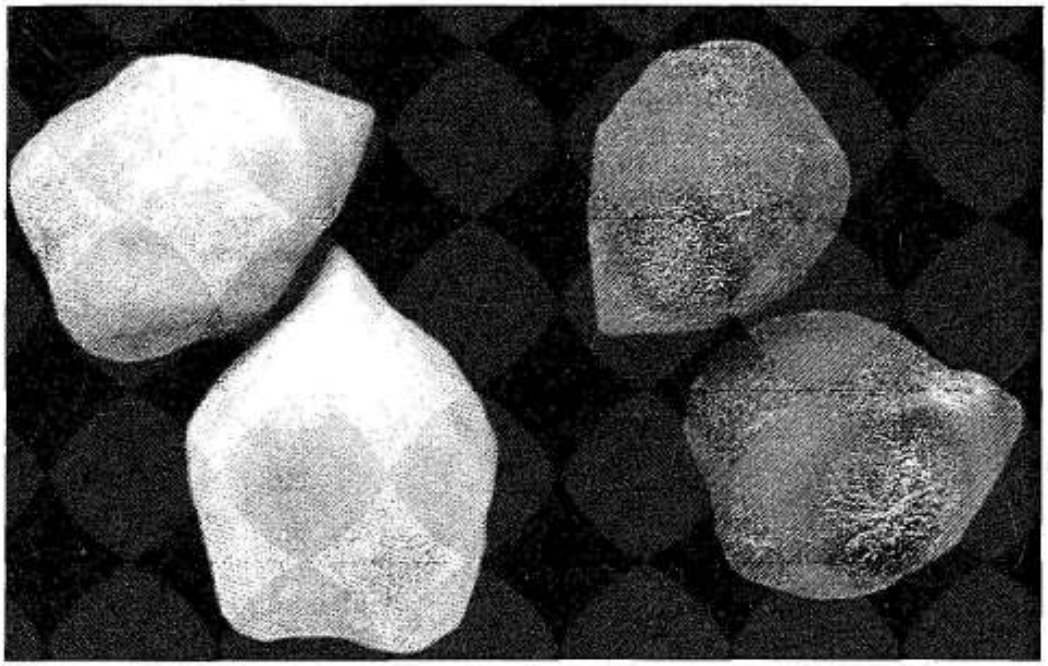


Figure 27A.
Dry seeds of chickpea showing damage caused by Ascochyta rabiei *(right); and healthy seeds (left), ×6.*

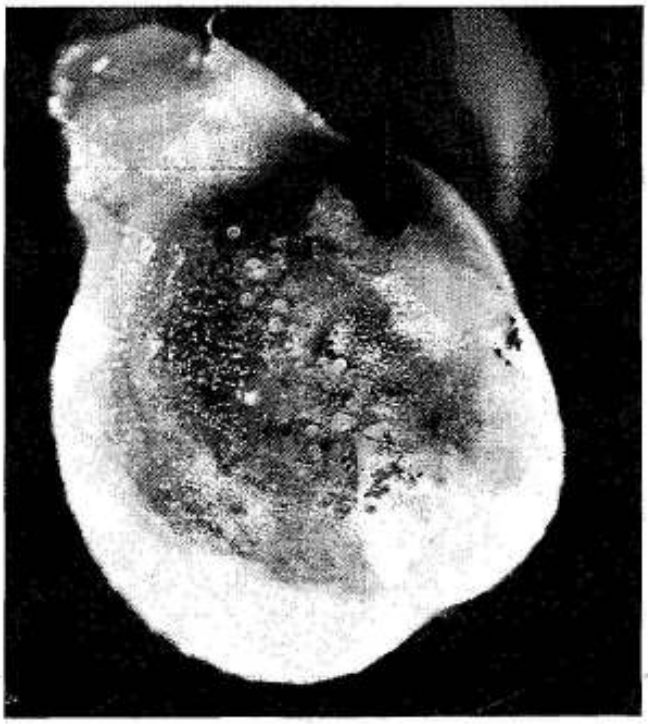


Figure 27B.
Infected chickpea seed after incubation showing the growth of A. rabiei, *×13.*

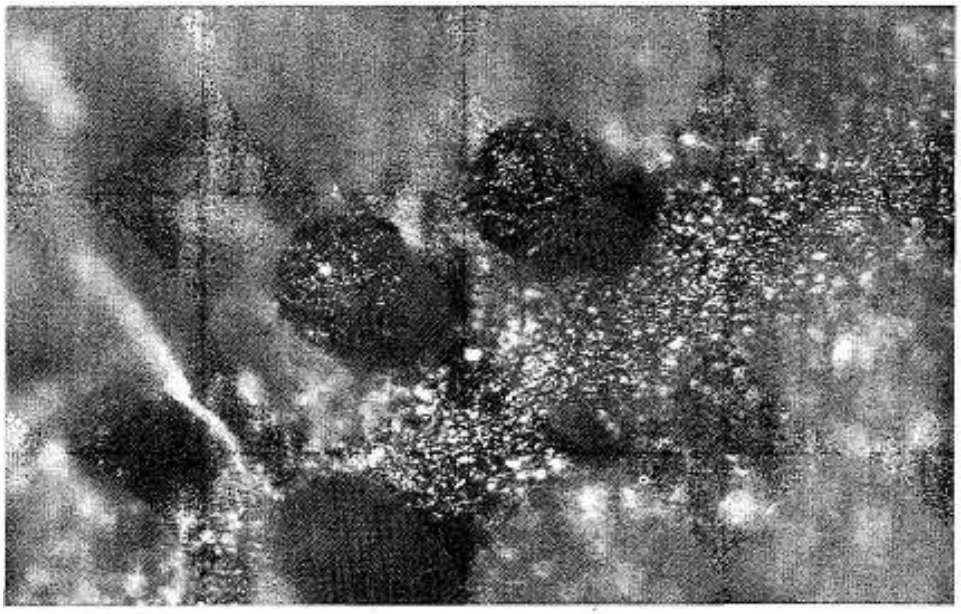


Figure 27C.
Pycnidia of A. rabiei, *×113.*

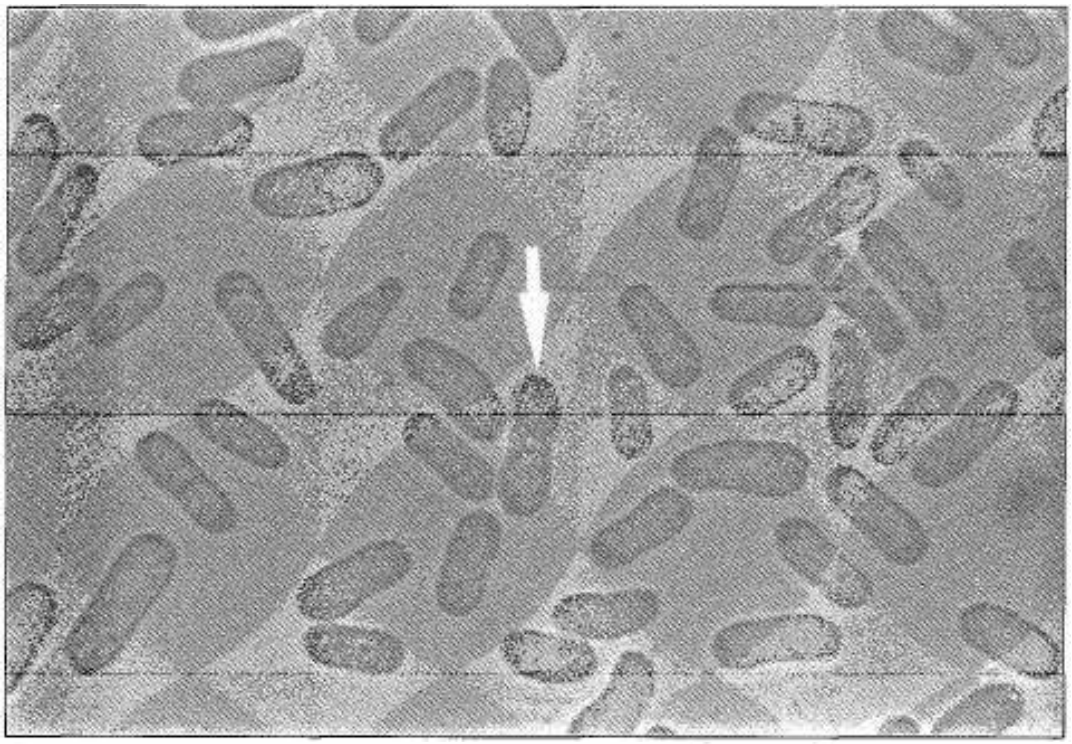


Figure 27D.
Bicelled conidia (arrowed) of A. rabiei, *×1130.*

Botrytis cinerea Pers. ex Fries

Botrytis cinerea causes gray mold/blight diseases of chickpea and groundnut. Gray mold of chickpea is an economically important disease. Infected seeds can be detected by visual examination and incubation tests. Infected dry seed appear discolored, shrunken, and mummified (Fig. 28A). Grayish white mycelium is sometimes seen on the seed surface. Sclerotia may also occur as admixtures in seed lots from infected fields. These sclerotia are gray-black, and irregular in shape. On incubated seed, the fungal growth consists of long, slender, and erect conidiophores which are branched at the apex. The fungus sporulates profusely, and superficially the growth resembles that of *Cladosporium* sp. However, colonies of *B. cinerea* are white to light gray to grayish brown, spreading short distances around the infected seed (Fig. 28B). **Conidiophores** can be seen at the tips and at intervals along the hyphae, and appear to arise as pegs on the swollen ends of hyaline branches. They are light gray, nearly 2 mm long, 16–30 µm thick, and dichotomously branched (Fig. 28C). Conidiophores are brown at the base, becoming paler towards the apex, with the ends of the branches often colorless, and bearing conidia in clusters (Fig. 28Da). **Conidia** are produced on short denticles at the tips of the conidiophores. They are usually single-celled, occasionally ellipsoidal or obovoidal, apiculate at the base, colorless to pale brown, and 4–20 × 4–18 µm (Fig. 28Db). The ornamented arrangement of conidia on the conidiophores resembles a cruciferous inflorescence under a stereobinocular microscope (Ellis and Waller 1974; Laha and Grewal 1983; Nene and Reddy 1987).

Control. Avoid collecting seed from endemic areas, and conduct pre-export crop health inspections. Seed treatment with carbendazim alone, or in combination with thiram at about 2.5 g kg^{-1} (Bavistin 25WP® + Thiram 50WP®) is recommended to eradicate the fungus from the seeds (Grewal 1982). Vinclozolin (Ronilan® at about 1 g kg^{-1} seed) eliminates internal and external infection on chickpea (Grewal and Laha 1983).

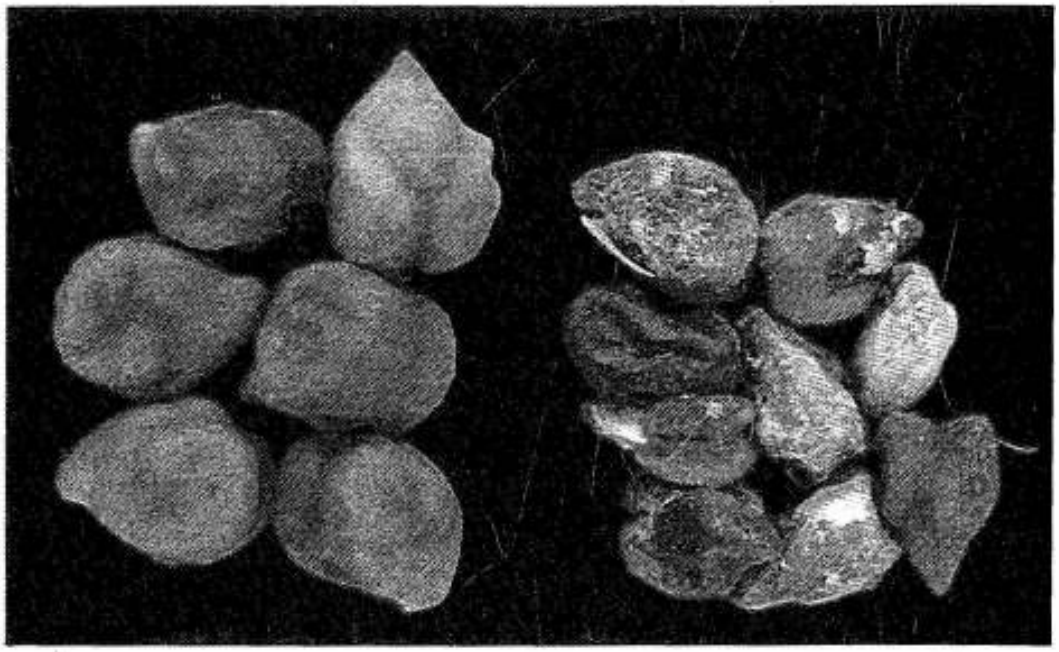


Figure 28A.
Dry seeds of chickpea showing damage caused by Botrytis cinerea *(right), and healthy seeds (left), ×4.*

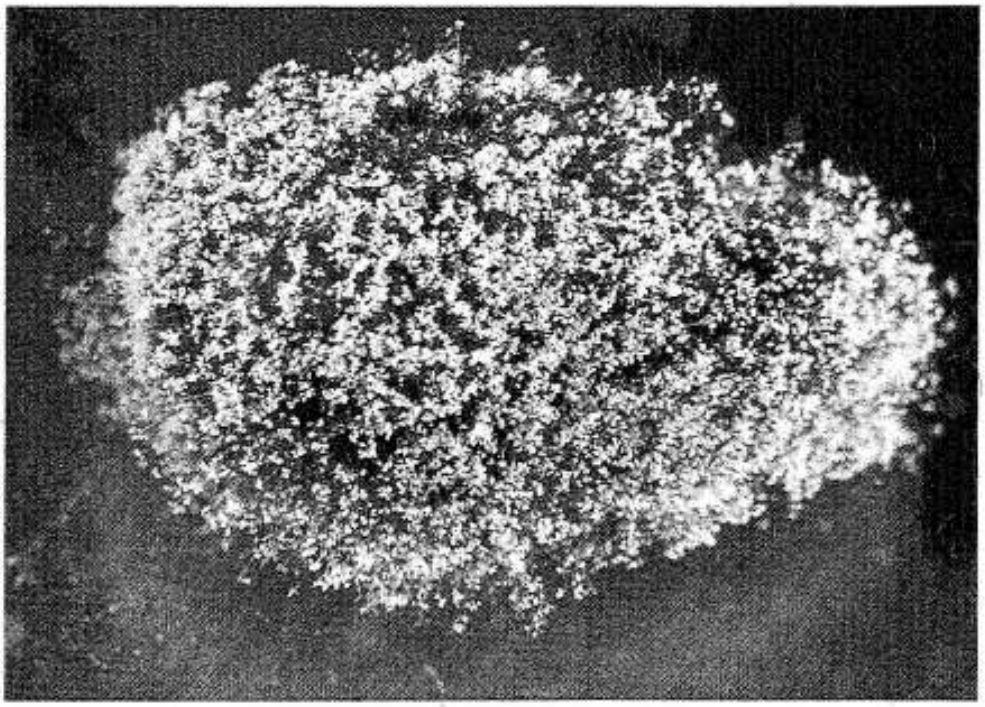


Figure 28B.
Infected chickpea seed after incubation, showing the growth of B. cineria, *×15.*

Figure 28C.
Dichotomously branched conidiophores bearing conidia of Botrytis cinerea *on chickpea seed, ×29.*

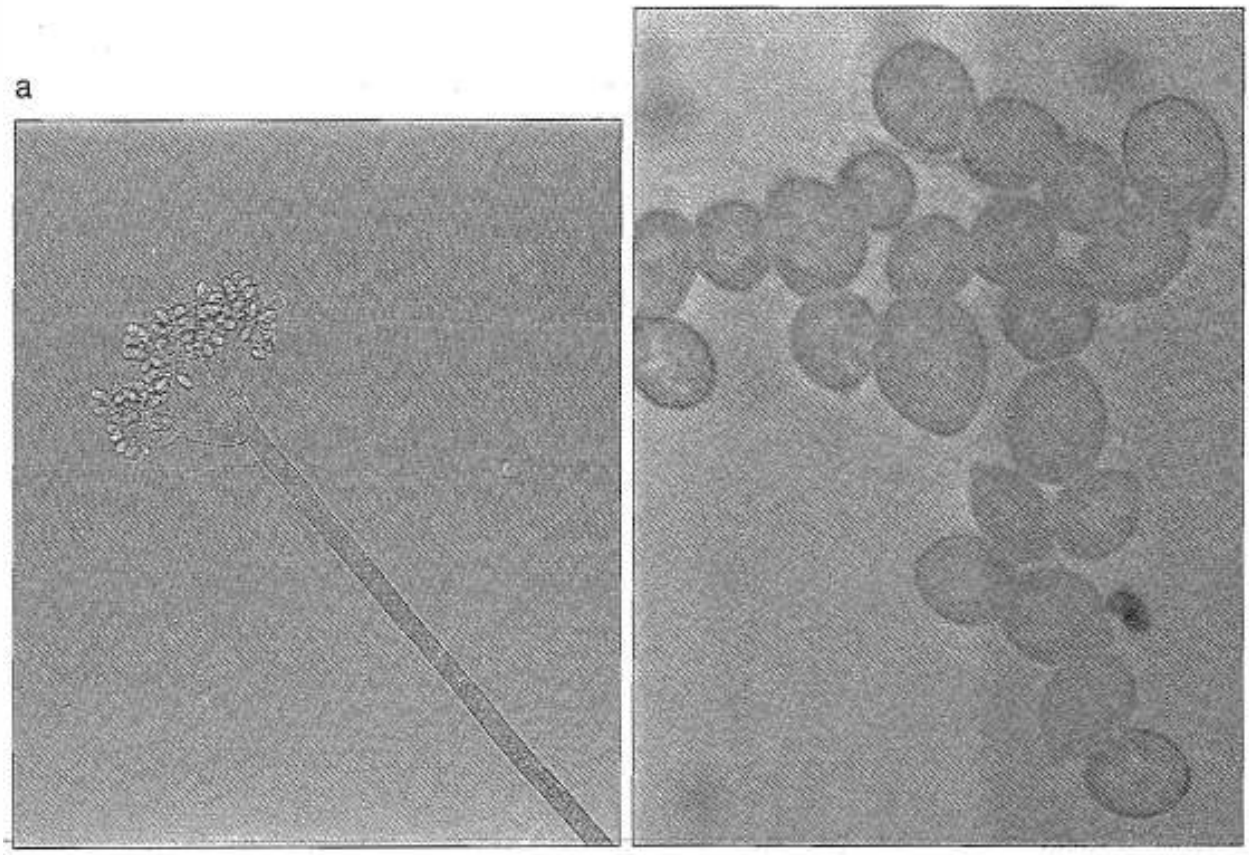


Figure 28D.
(a) Conidiophore, ×451, and (b) conidia, ×1130 of B. cinerea.

Colletotrichum dematium (Pers. ex Fr.) Grove

Colletotrichum dematium causes anthracnose in chickpea and groundnut, the disease can become serious in warm and humid weather. Infected seeds can be detected by visual examination and incubation tests. Dry infected seeds of chickpea appear shriveled, discolored (Fig. 29Ab), and sometimes the fungus can be seen as acervuli with black setae (Fig. 29Aa). At times only the white mycelium is seen on profusely infected seed. On incubated seed, the fungus produces circular, erumpent, dark brown to black acervuli (Figs. 29Ba and b). These **acervuli** are scattered throughout the seed or aggregated (confluent), and in groups. Acervuli are usually longitudinally elongated, at first covered by the cuticle and epidermis, but later become strongly erumpent. They exude spores in pale, smoke-gray masses. The color of the acervular mass ranges from pale to bright orange. Within this acervular mass, numerous thick black, erect and hair-like spines or setae can be seen (Fig. 29C). **Conidia** are hyaline, single-celled, measure 2.5–4 × 15–32 μm, fusoid, and bluntly tapered at both ends, with acute apices and truncate bases. Conidia are irregularly guttulate and smooth-walled (Figs. 29Da and b). **Setae** are longer than the acervular mass. They are 1–5 septate, swollen at the base, 150–300 μm long, straight, rarely curved, irregular at the apex, and with thick smooth walls (Mishra et al. 1975; Ghewande et al. 1987; Nene and Reddy 1987).

Control. Chickpea seeds intended for export should be collected from disease-free areas or fields inspected during the active growth phase. Information on seed treatment to eradicate seedborne inoculum is not available.

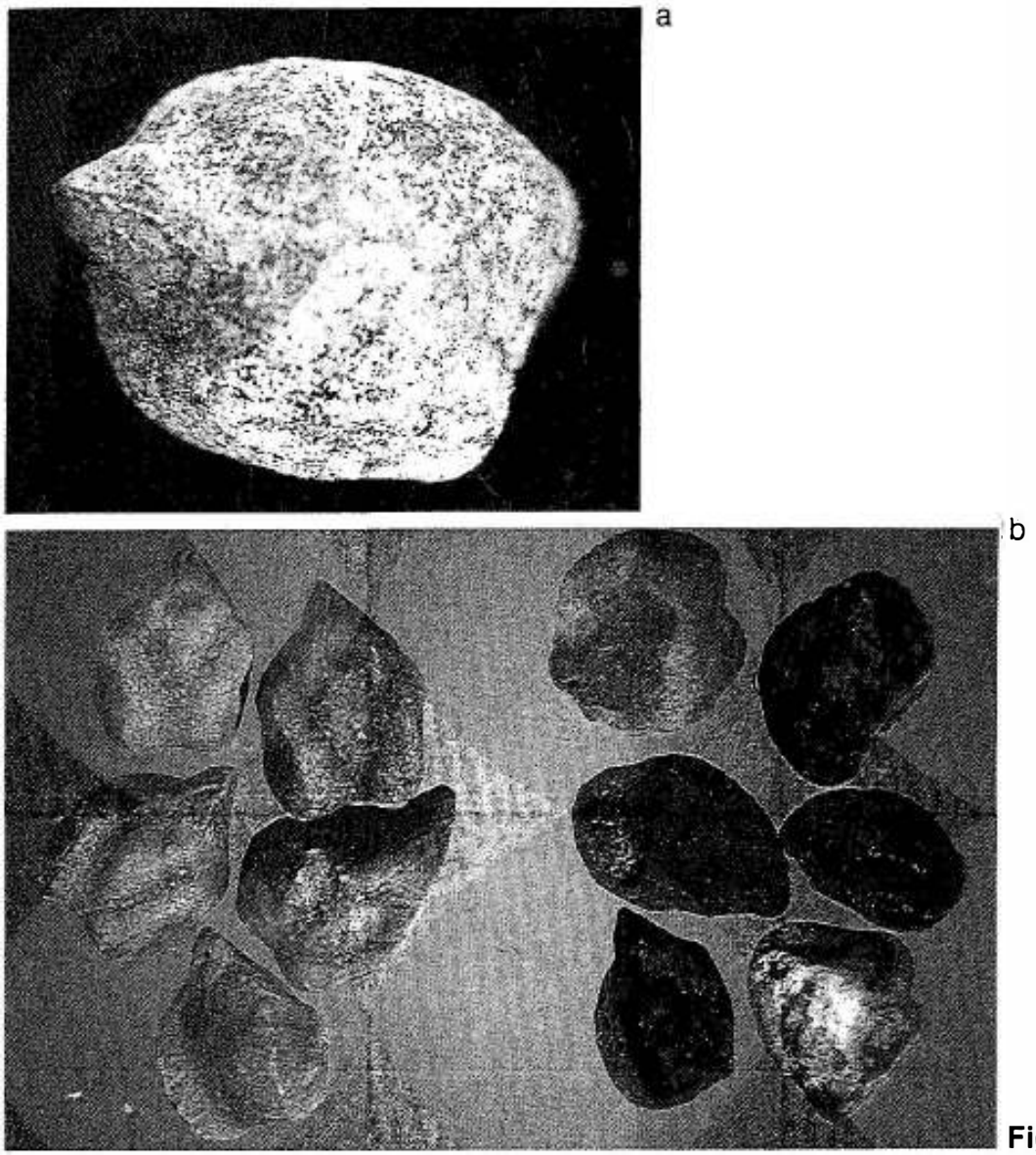


Figure 29A.
(a) Dry acervuli on chickpea seed, ×9, and (b) dry chickpea seeds showing damage caused by Colletotrichum dematium *(right), and healthy seeds (left), ×4.*

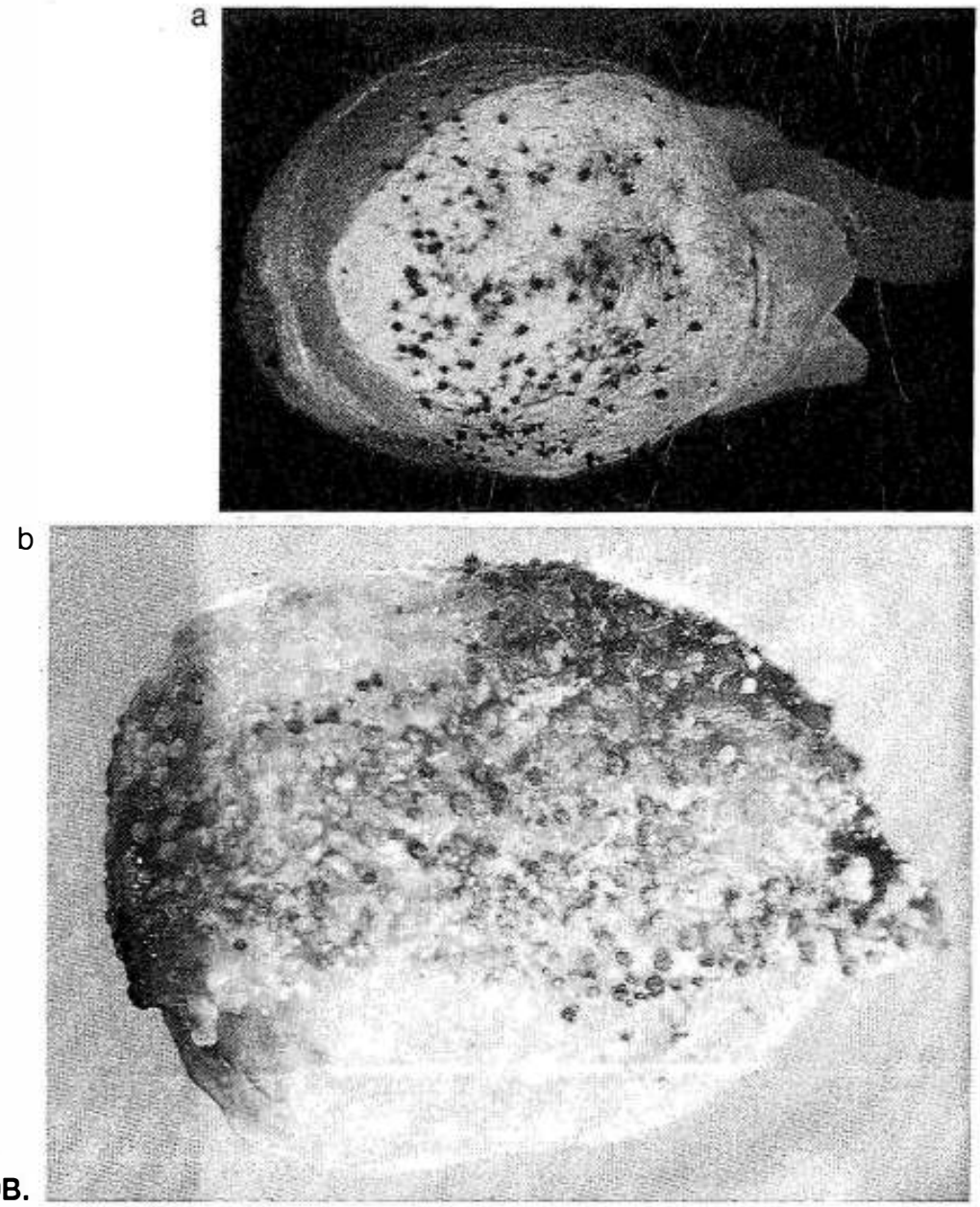


Figure 29B.
Seeds of: (a) chickpea, ×13, and (b) groundnut, ×20, after incubation, showing acervuli of C. dematium.

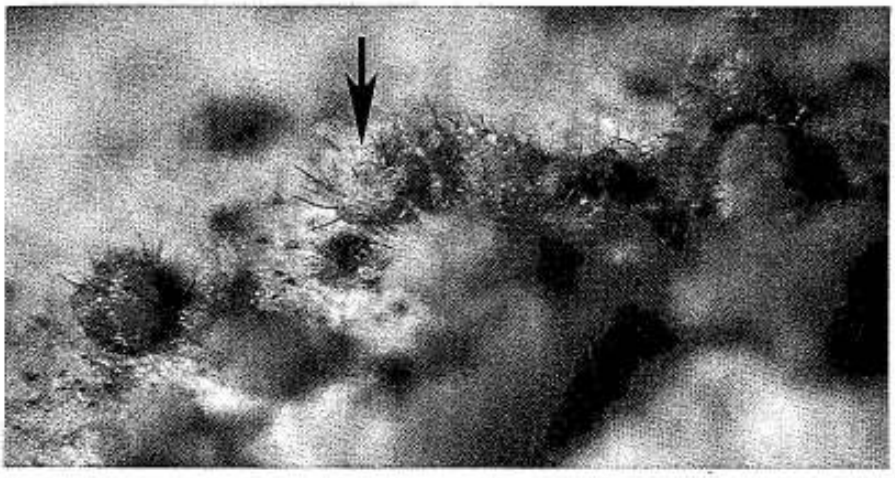
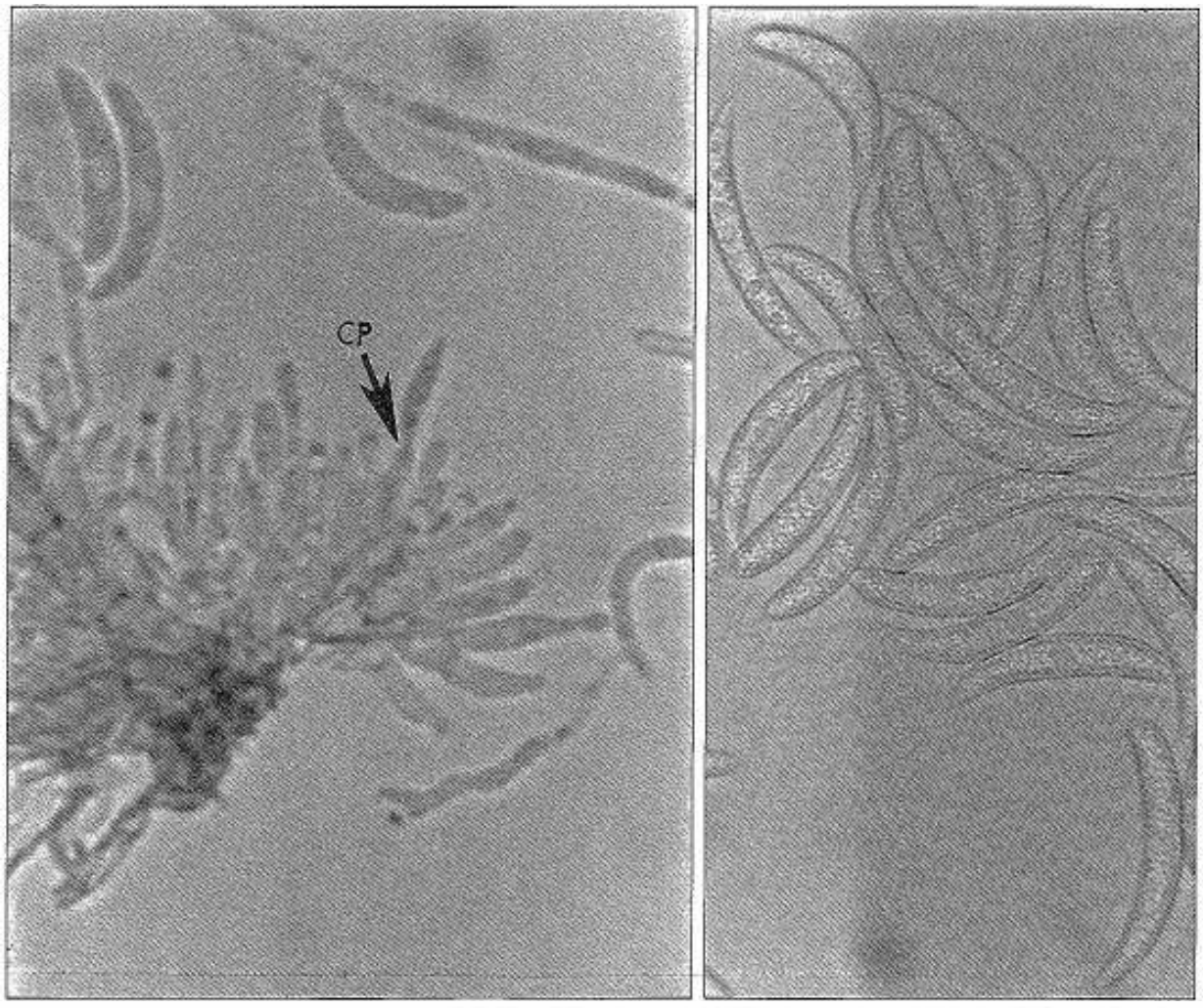


Figure 29C.
Acervuli (arrowed) and setae of C. dematium *on groundnut seed, ×113.*

a



b

Figure 29D.
(a) Conidiophores (CP), ×1130, and (b) conidia of C. dematium, *×1130.*

Fusarium oxysporum Schlechtend emend. Snyd. & Hans. f. sp. *ciceri* (Padwick) Snyd. & Hans.

This fungus causes vascular wilt of chickpea, one of the economically important diseases affecting this crop. Infected seeds can be detected by visual examination and incubation tests. Dry, infected seeds are discolored, small, and wrinkled (Fig.30A), but healthy-looking seed may also harbor the pathogen. The fungus produces white to light orange, aerial mycelium and sporodochia on incubated seed. The **mycelium** is profusely branched, covers the entire seed, and is white to light pink. **Sporodochia** are rarely produced, but if present, are completely covered by the aerial mycelium (Fig.30B). **Microconidia** are abundant, and are produced on short, unbranched monophialides (microconidiophores) in small, dry, false heads (Figs.30C and Da). Microconidia are hyaline, single-celled, oval to cylindrical, straight to slightly curved, and 2.5–3.5 × 5–11 μm (Fig.30Db). **Macroconidia** are sparse and produced on branched macroconidiophores (Figs. 30Dc and d). They are fusoid with pointed ends, hyaline, have 3–5 septa, and measure 3.5–4.5 × 25–65 μm. **Chlamydospores** are usually intercalary and are produced singly or in pairs. They are globose to subglobose, thick-walled, and smooth-surfaced (Fig.30De). Chlamydospore-like swellings are often seen on the hyphae (Haware et al. 1986; Nene and Reddy 1987).

Control. Collect seed lots from disease-free fields. Discard infected seeds during visual examination. Select healthy-looking seeds and treat them with a (1:1) mixture of 30% benomyl + 30% thiram (Benlate T® at about 1.5 g kg^{-1}) this will eradicate the seedborne infection of the fungus (Haware et al. 1978).

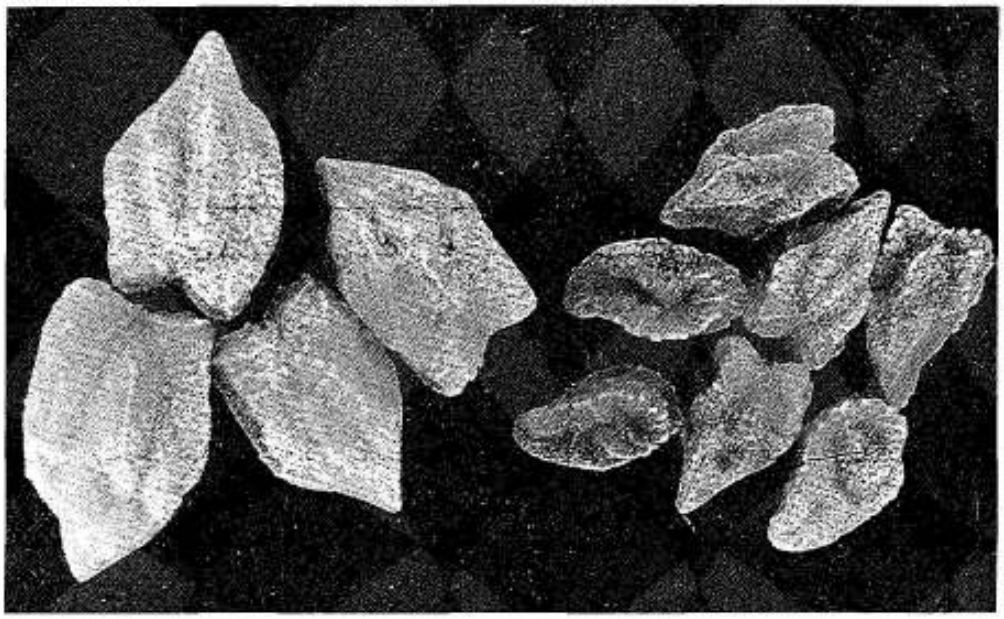


Figure 30A.
Dry infected seeds of chickpea showing damage caused by Fusarium oxysporum *f. sp.* ciceri *(right), and healthy seeds (left), ×14.*

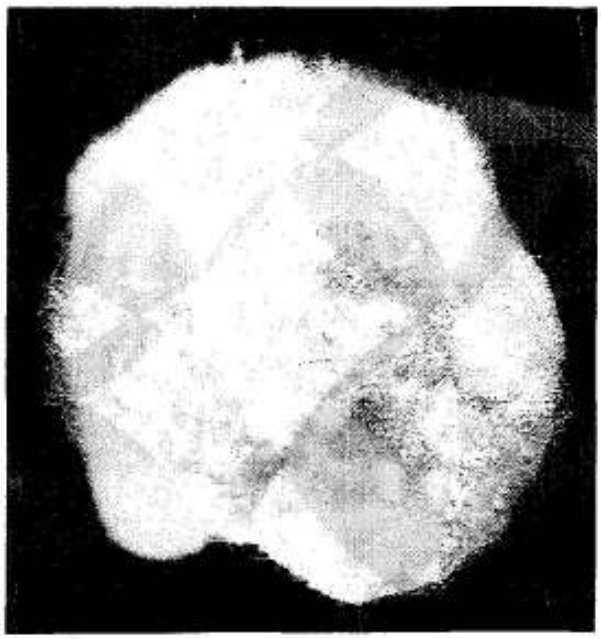


Figure 30B.
Infected chickpea seed after incubation, showing sporodochia covered by aerial mycelium F. oxysporum *f. sp.* ciceri, *×13.*

Figure 30C.
False heads of F. oxysporum *f. sp.* ciceri *(arrowed) in which microconidia are formed on the aerial mycelium, ×113.*

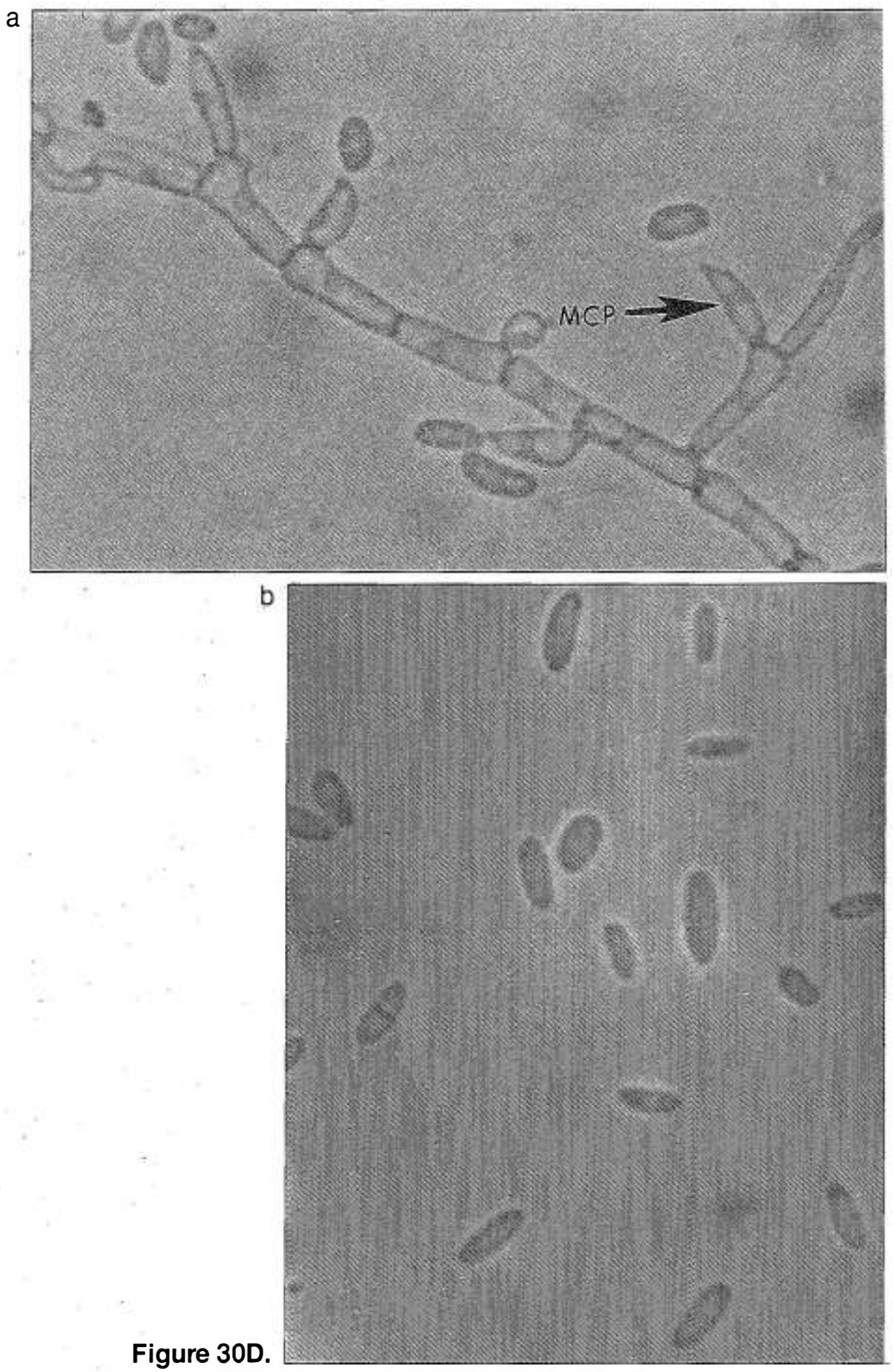


Figure 30D.
(a) Microconidiophores (MCP), ×1130, (b) microconidia, ×1130,

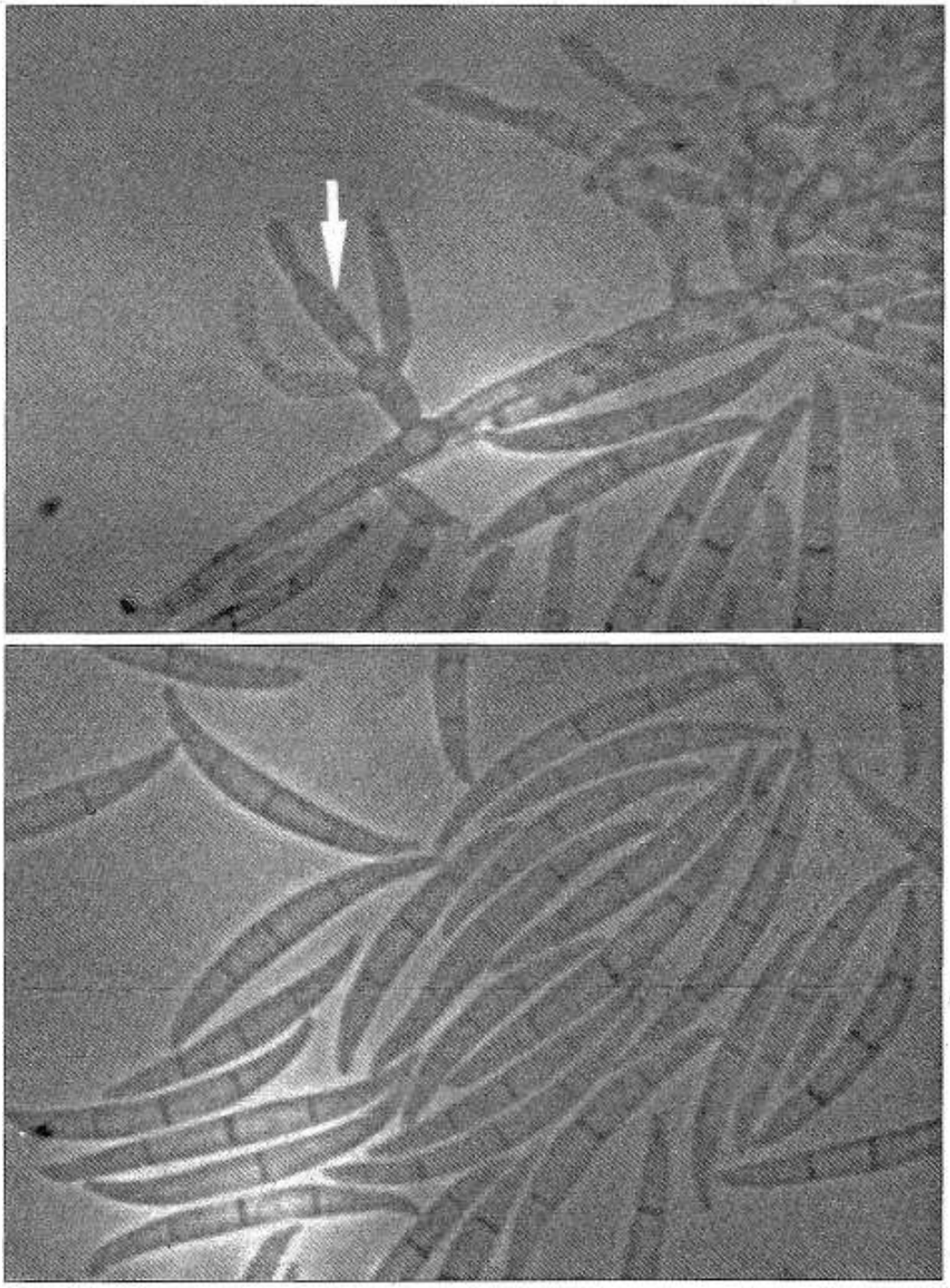


Figure 30D.
(c) macroconidiophores (arrowed), ×1130, (d) macroconidia, ×1130, and

e

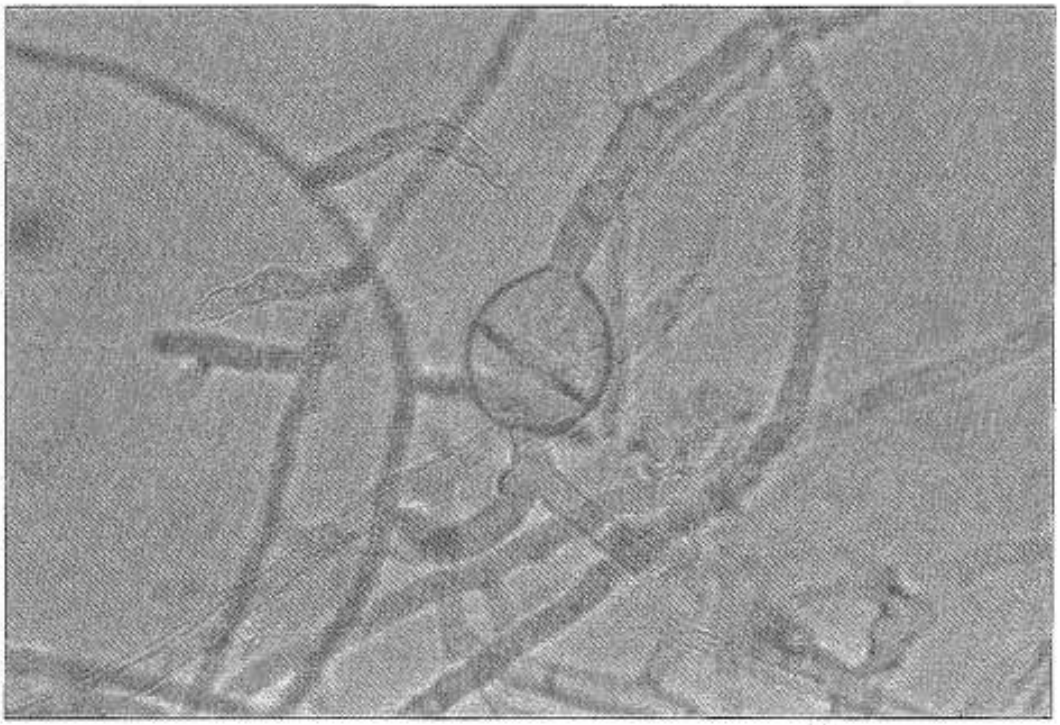


Figure 30D.
(e) chlamydospores, of F. oxysporum *f. sp.* ciceri, *×1130.*

Fusarium solani (Mart.) Sacc.

Fusarium solani is a pathogen of chickpea and groundnut, and causes root rot and wilt diseases. Seeds are infected through the soil or through their mother plants in the field. Seed infection can be detected by incubation tests. The fungus produces abundant loose, aerial, white mycelium on incubated seed (Fig.31A). In this **mycelium**, several shiny, hyaline, and transparent to milky-white spherical droplets can be seen hanging at the tips of long thin stalks. These stalks are primary **conidiophores**, which arise laterally from the hyphae in the aerial mycelium. The hanging droplets are moist false heads in which conidia are produced (Figs.31Ba and b). Mucoid and moist pionnotes and snow-white to dull white **sporodochia** are also produced on the seed surface (Fig.31Ca). **Microconidiophores** (monophialides) that bear the microconidia are very long and slender, and measure 15–40 × 2–3 μm (Fig.31Cb), whereas those bearing macroconidia (macroconidiophores) are short and measure 10–25 × 3–4.5 μm (Fig.31Cc). **Microconidia** are hyaline, 1–2 septate, oval, ellipsoid to subcylindrical, and measure 5–20 × 2.8–7 μm (Fig.31Cd). **Macroconidia** are hyaline, stout, measure 22–75 × 3.5–7 μm, subcylindrical or slightly curved, with short blunt and rounded apical cells and indistinctly pedicillate basal cells. The walls of the conidia are thick, with dorsal and ventral surfaces parallel for most of their length. They are mostly 3–septate but 4–7 septate conidia are not uncommon (Fig.31Ce). **Chlamydospores** are formed singly and in pairs, or in clusters in sporodochia. They are globose to subglobose, smooth or rough-walled, and 6–11 μm in diameter (Fig.31Cf) (Kraft 1969; Ram Nath et al. 1970; Booth 1971).

Control. Seed treatment with benomyl (Benlate®) at about 2 g kg^{-1} is effective to reduce the seedborne inoculum of the fungus (Vishwakarma and Basu Chaudhary 1982).

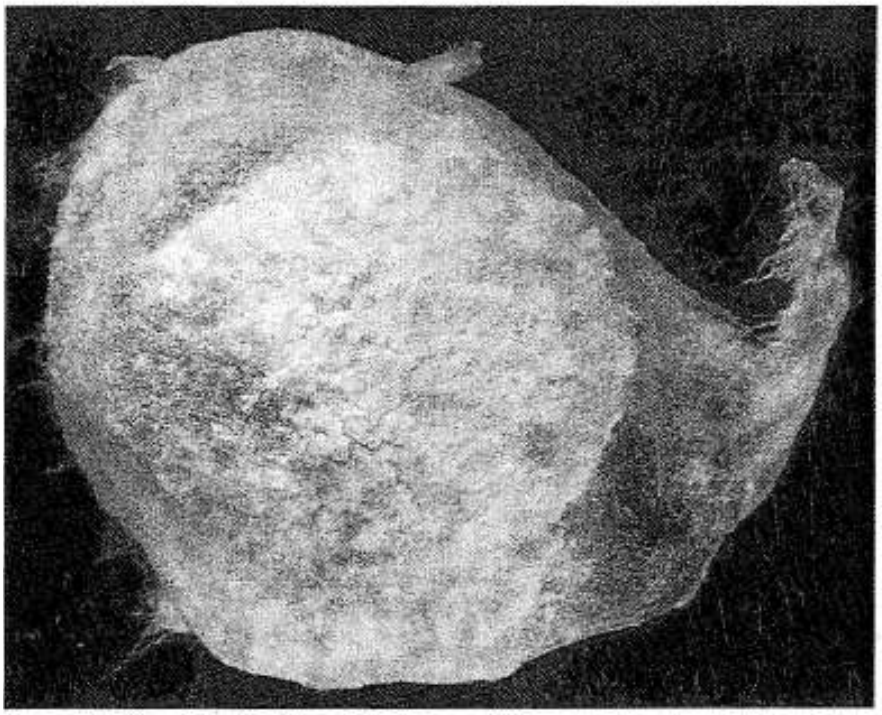


Figure 31A.
Infected chickpea seed after incubation, showing the growth of Fusarium solani, *×16.*

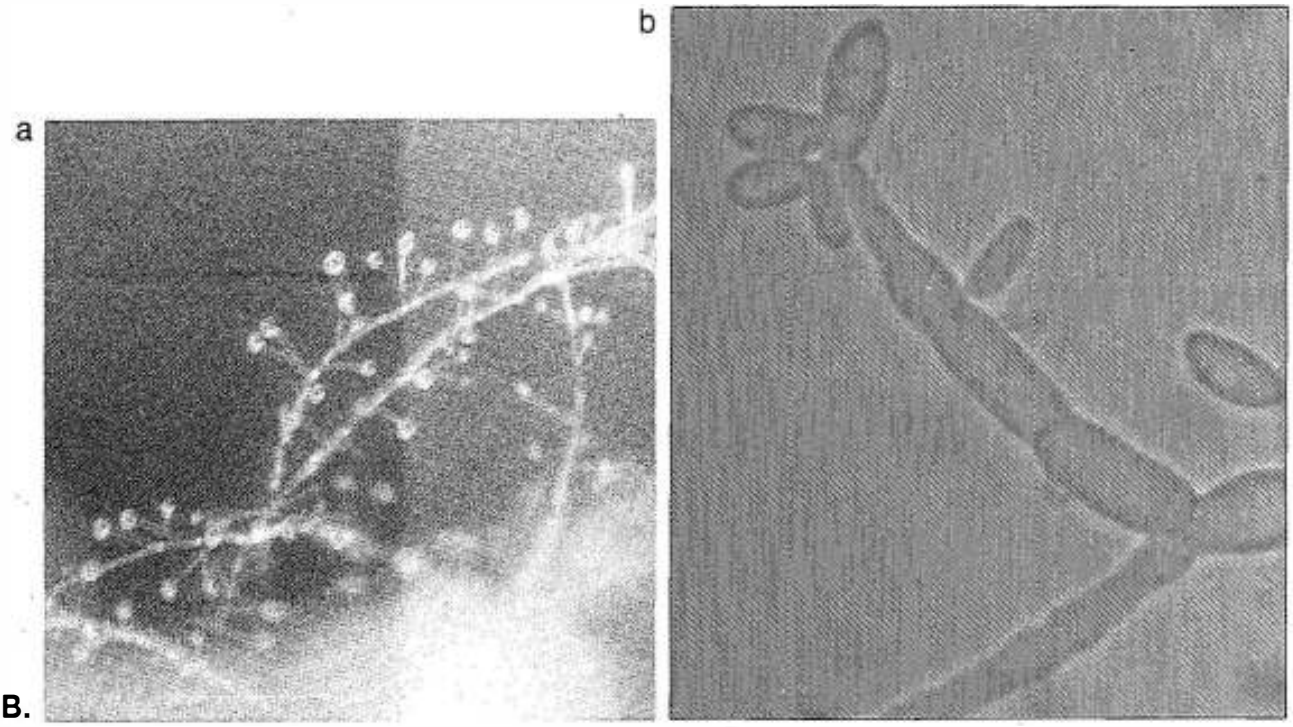


Figure 31B.
(a) False heads of conidia produced on the aerial mycelium, ×60, and (b) false head of F. solani, *×1130.*

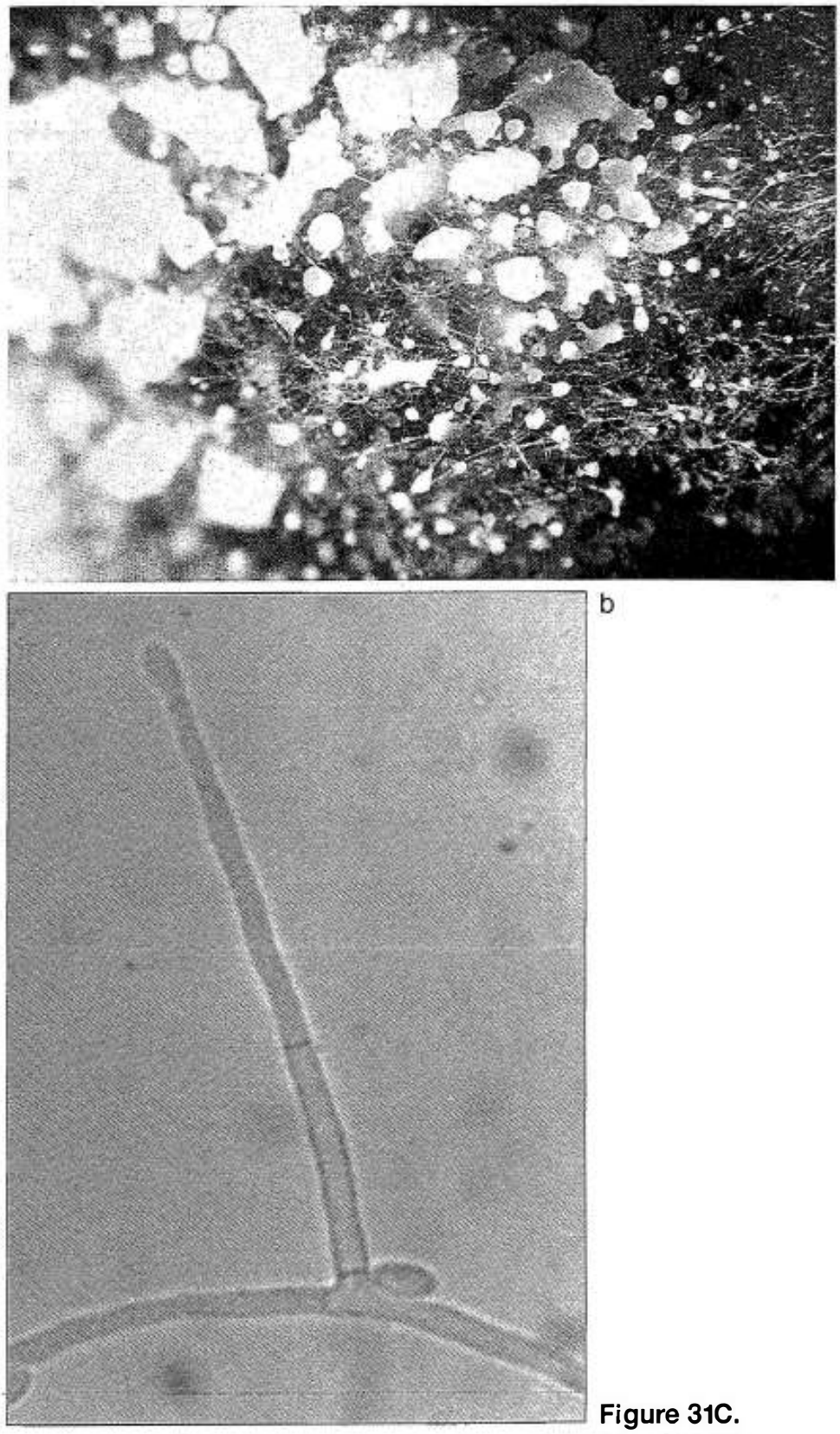


Figure 31C.
(a) Sporodochia, ×26, (b) microconidiophore, ×1130,

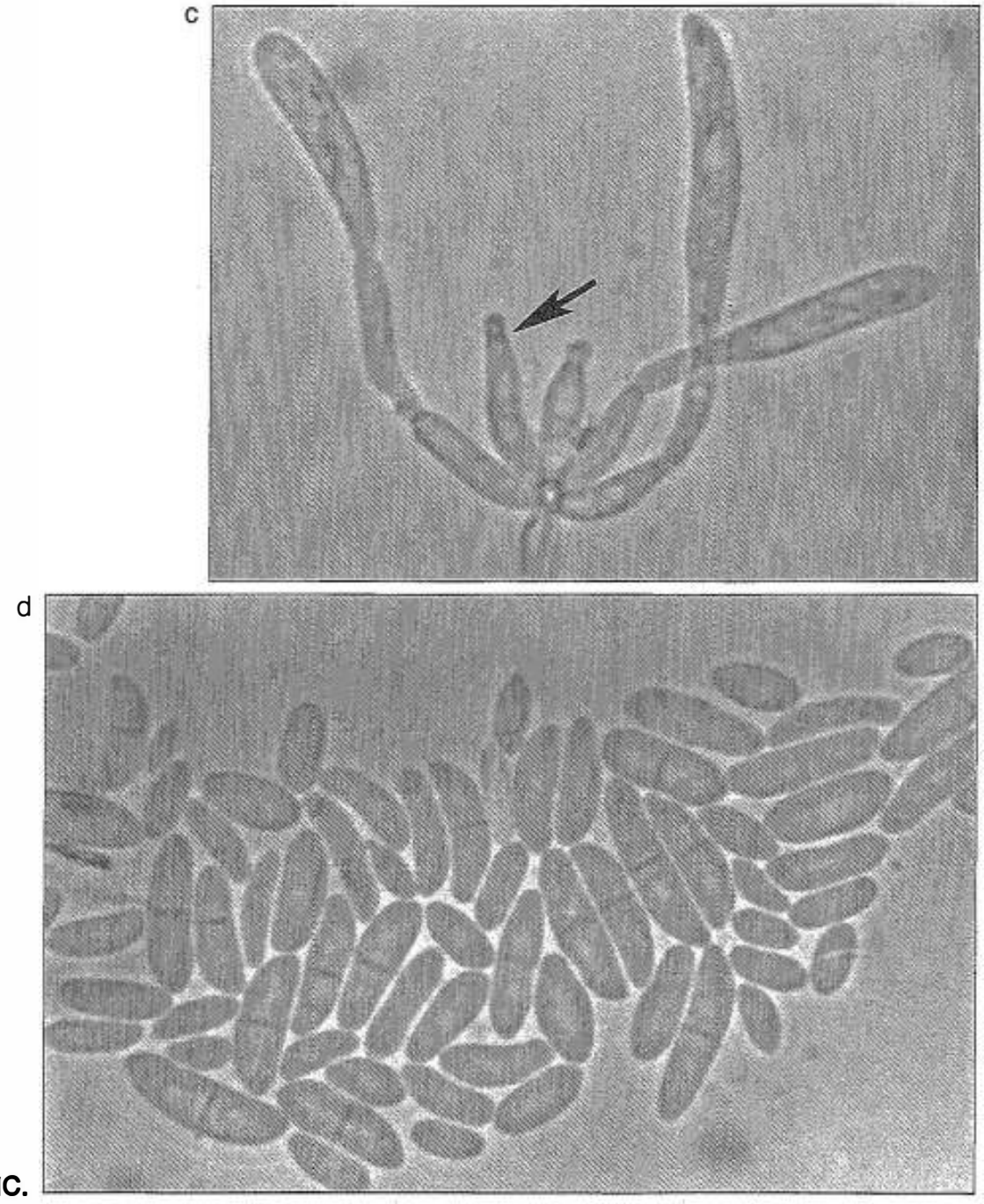
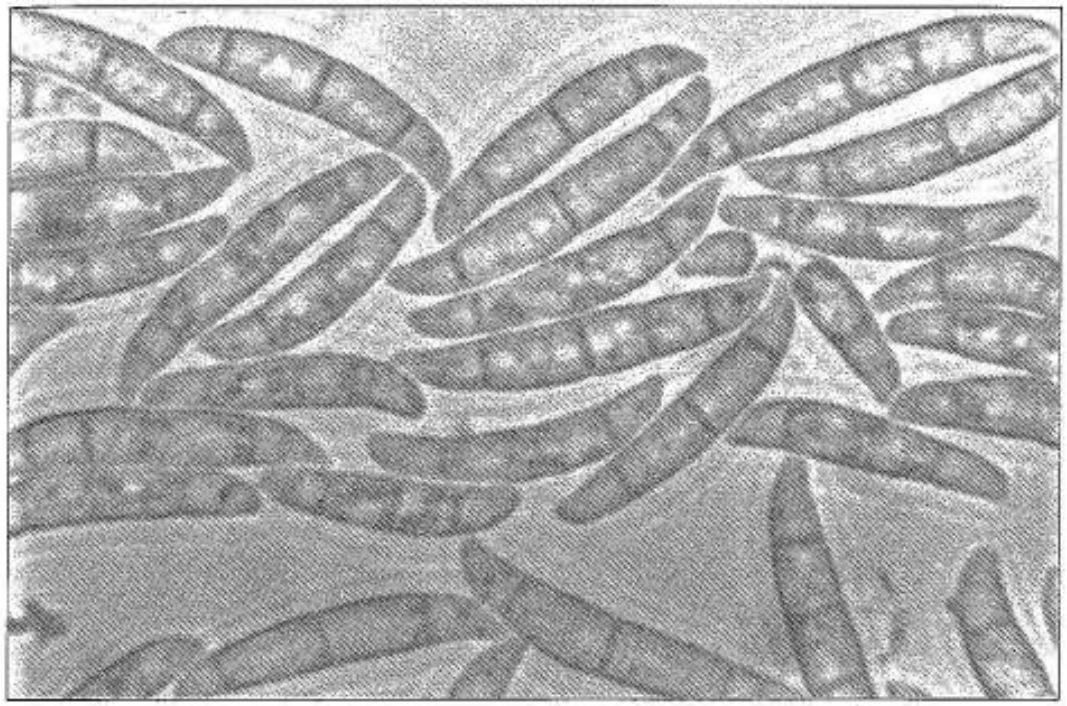


Figure 31C.
(c) macroconidiophores (arrowed), ×1130, (d) microconidia, ×1130,



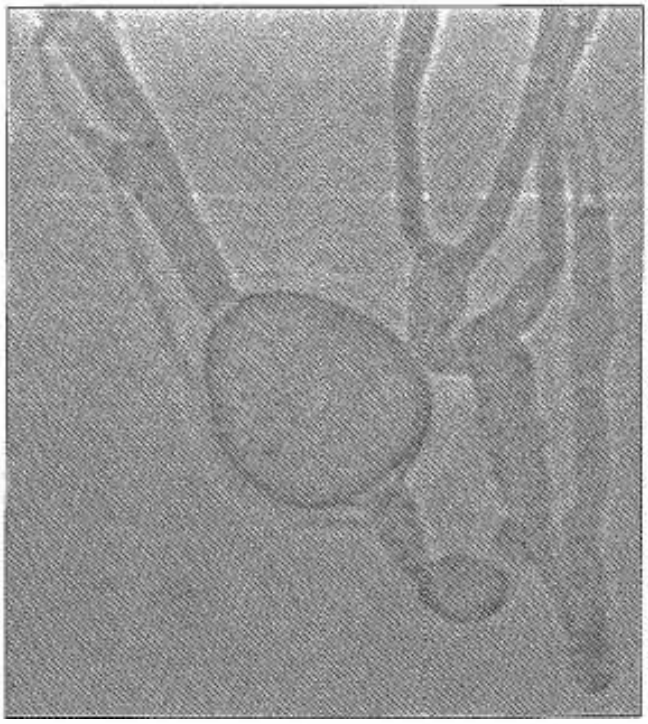


Figure 31C.
(e) macroconidia, ×1130, and (f) chlamydospores of F. solani, *×1130.*

Neocosmospora vasinfecta E.F.Smith

Neocosmospora vasinfecta is known to cause minor diseases by attacking the roots of chickpea, pigeonpea, and groundnut plants. Seedborne infection can be detected by incubation tests. The conidial state of the fungus, which belongs to the genus *Acremonium*, is usually observed on seed after 7 days of incubation (Fig. 32A). The growth superficially resembles that of fungi belonging to the genus *Fusarium* (Figs. 32Ba, 32Ca and b). However, when such seeds are incubated for more than 7 days (up to 14 days), perithecia are formed by the fungus, and can be seen under a stereobinocular microscope. After 7 days incubation abundant, bright orange perithecia are seen growing superficially throughout the seed surface, either singly or in a confluent mass. The **mycelium** is profuse, hyaline, septate, and branched, with hyphal walls. **Perithecia** (Fig. 32Bb) are superficial, globose to obpyriform, red orange or occasionally brown, the ostiolar neck is short, 300–500 μm long, 280–480 μm in diameter, and lined with paraphyses. The outer cells of the perithecium are bright orange, and its inner cells are hyaline. **Asci** are unitunicate, cylindrical or rarely clavate, short-stalked, 8–15 μm long, apex undifferentiated or with an indistinct nonamyloid ring, and measure 80–95 × 10.5–15 μm (Fig. 32Cc). There are eight ascospores in each ascus. **Ascospores** are aseptate, monostichous, globose, brown, with a distinct wrinkled epispore. They are usually uniseriate, buff to solomon pink when in a mass, pale yellow individually, and measure 10.5–15.5 × 7.5–12 μm (Fig. 32Cd). **Paraphyses** are hyaline, inconspicuous, loosely jointed, and unbranched (Udagawa 1963; Cannon and Hawksworth 1984; Haware and Nene 1976; Barbosa Maria 1965; Beard and Van Wyk 1985).

Control. Good crop husbandry will produce healthy seed and prevent seedborne infection. Information on fungicidal seed treatment to eradicate seedborne inoculum is not available.

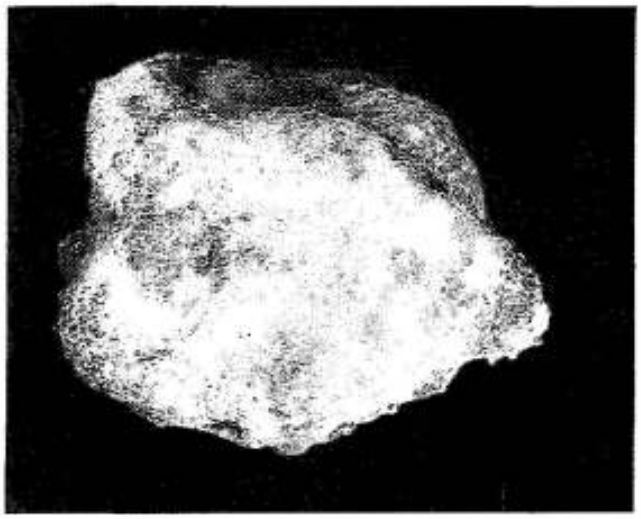
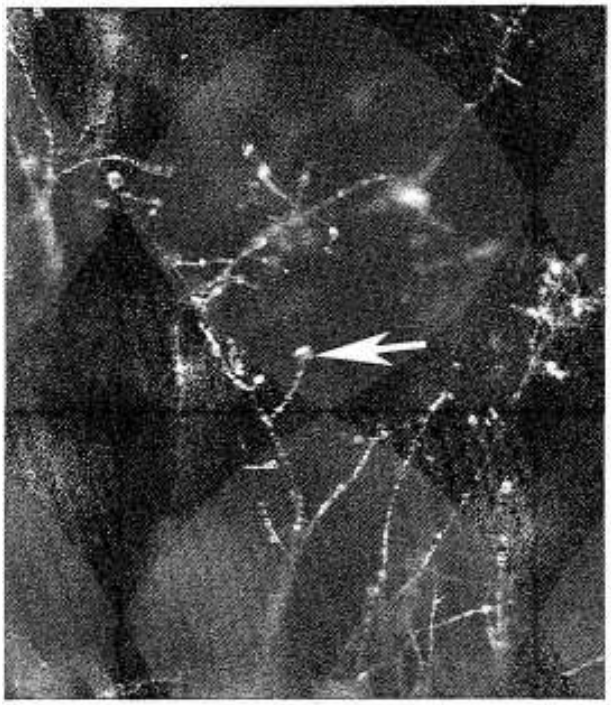


Figure 32A.
Infected chickpea seed after incubation, showing the growth of Neocosmospora vasinfecta. *White growth represents the imperfect state (*Acremonium *sp), and orange red the perfect state of the fungus, ×15.*

a



b

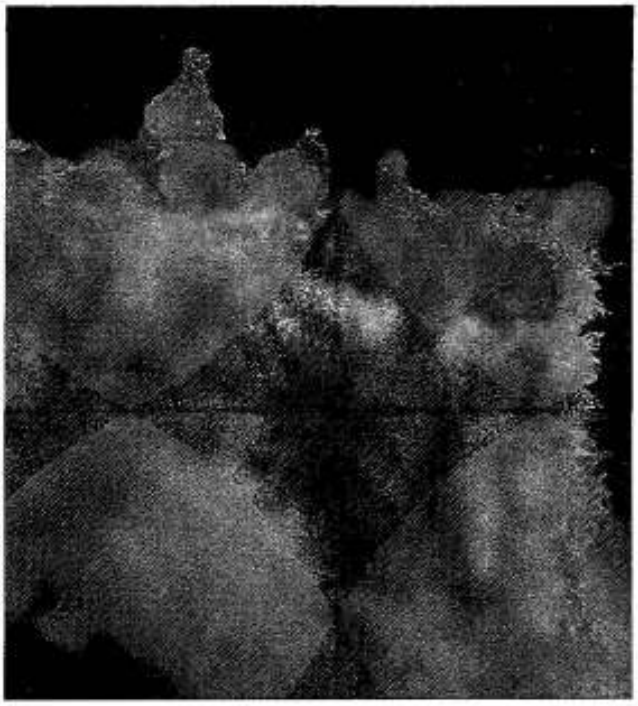


Figure 32B.
(a) Conidial heads (arrowed) produced on the aerial mycelium by Acremonium *sp, ×75, (b) perithecia of* N. vasinfecta, *×117.*

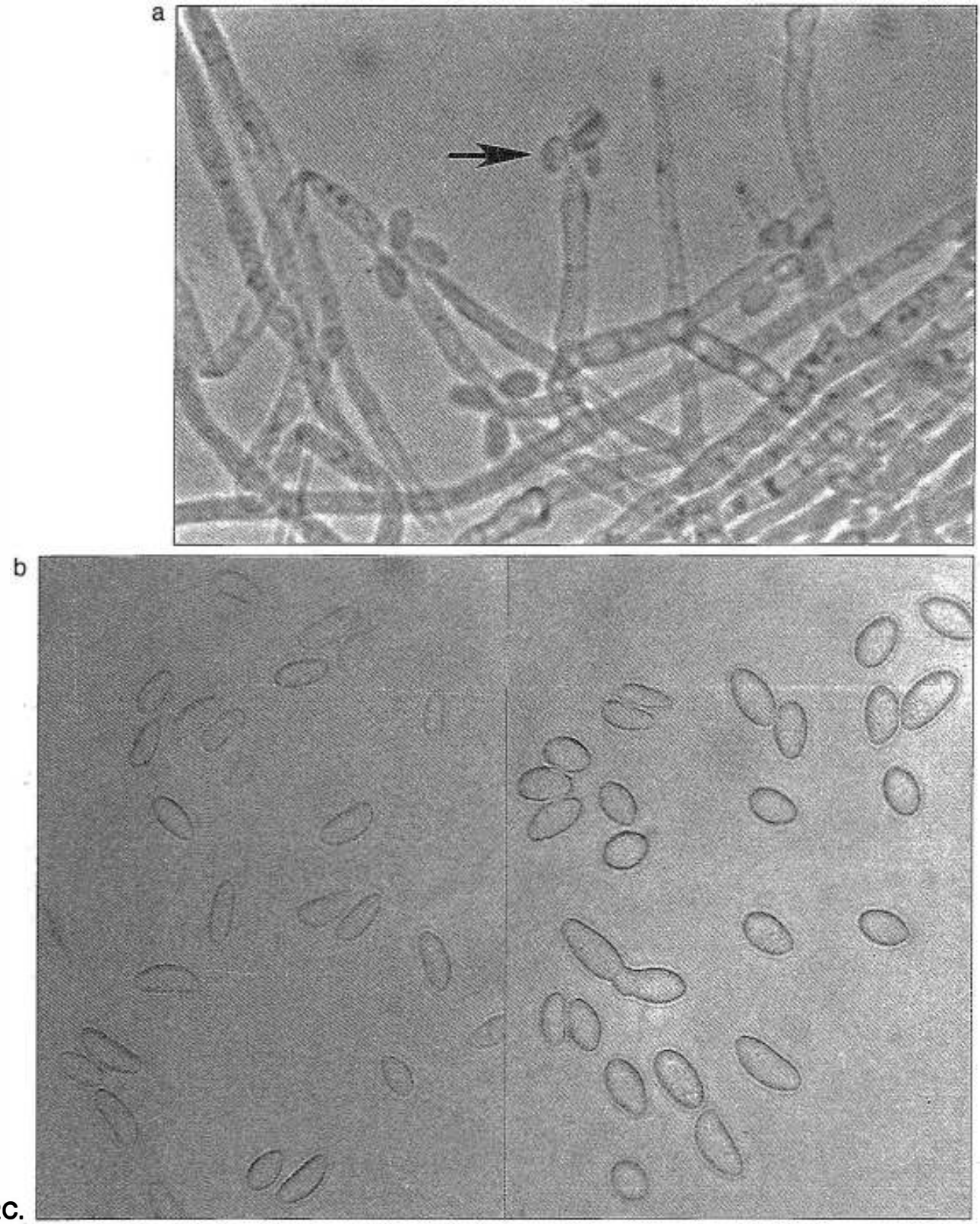


Figure 32C.
*(a) Conidial head (arrowed), ×1130, (b) conidia (*Acremonium sp), ×1130,

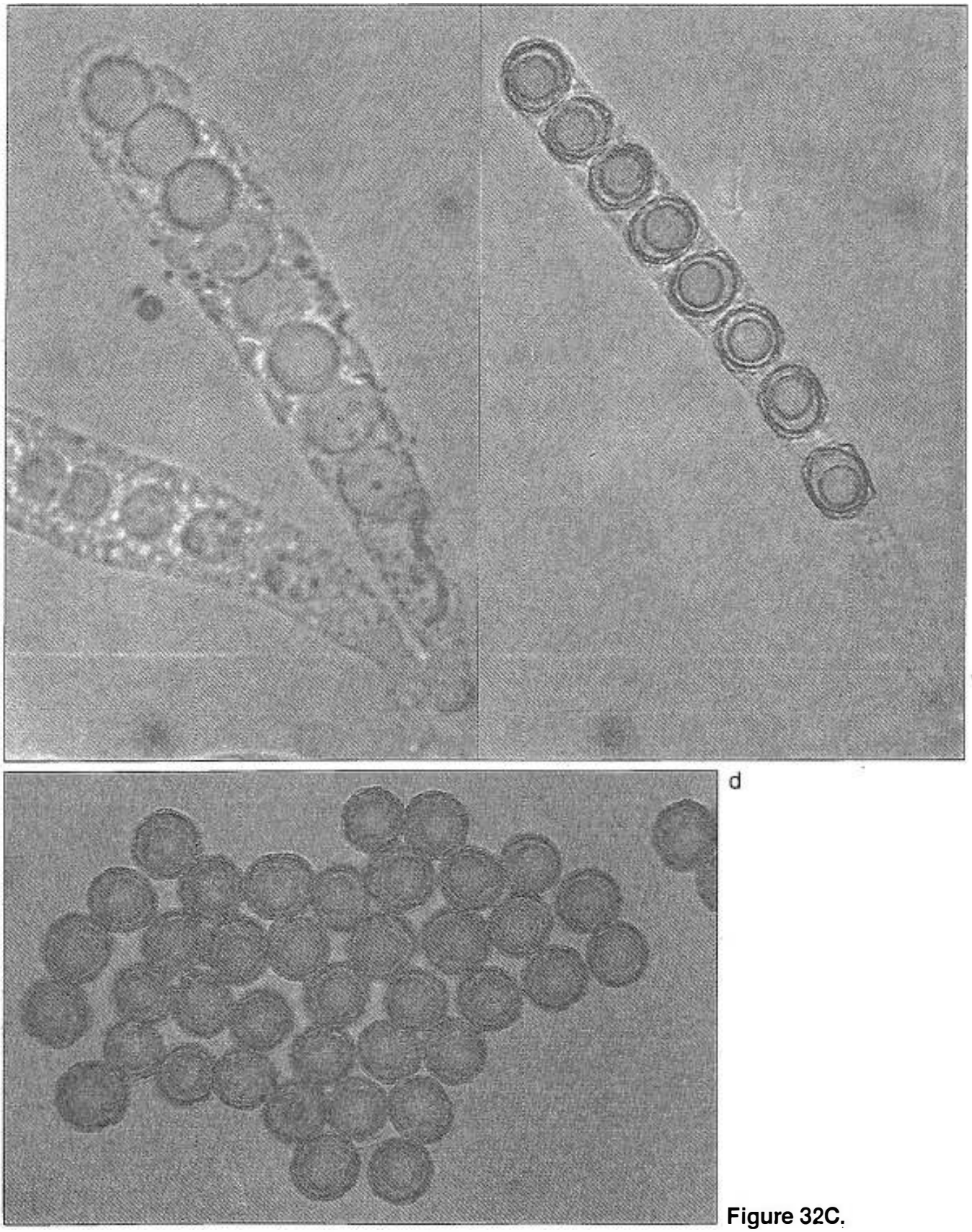


Figure 32C.
(c) asci, ×1130, (d) ascospores of N. vasinfecta, *×1130*

Phoma medicaginis Malbr. & Roum

Phoma medicaginis causes chickpea leaf spot, a seedborne disease. The fungus is similar in morphology to *P. medicaginis* Malbr. & Roum. var. *pinodella* (Jones) Boerema. *P. medicaginis* is usually carried as mycelium on the surface of the seed. Infected seeds can be detected by incubation tests. The fungus produces dark brown to black, shiny pycnidia scattered on the surface of incubated seed (Fig. 33Aa). The **mycelium** is dark gray to black. Hyphae are thick, branched, and have rough surfaces. **Pycnidia** arise singly or in aggregate masses with extremely variable shapes, 200–300 µm in diameter, usually globose to subglobose, becoming irregular in shape when produced in groups. Conidia ooze out of the pycnidia in a slimy, dark cream mass, which rests on the ostiole as a droplet (Fig. 33Ab). The **conidia** are hyaline to pale yellow, aseptate, straight or slightly irregular, and measure 4.5–8 × 2–3 µm. They may also be cylindrical, biguttulate or variably guttulate (Fig. 33B). **Chlamydospores** are small, thick, opaque, dark brown to black, spherical to irregular, and have smooth to rough surfaces. They may be terminal or intercalary, or formed in chains (Punithalingam and Gibson 1976; Haware and Nene 1981; Nene and Reddy 1987).

Control. Collecting chickpea seeds from disease-free fields is an important way to reduce the chance of disseminating the fungus to new areas. No information is available on the fungicidal control of seedborne inoculum of this fungus.

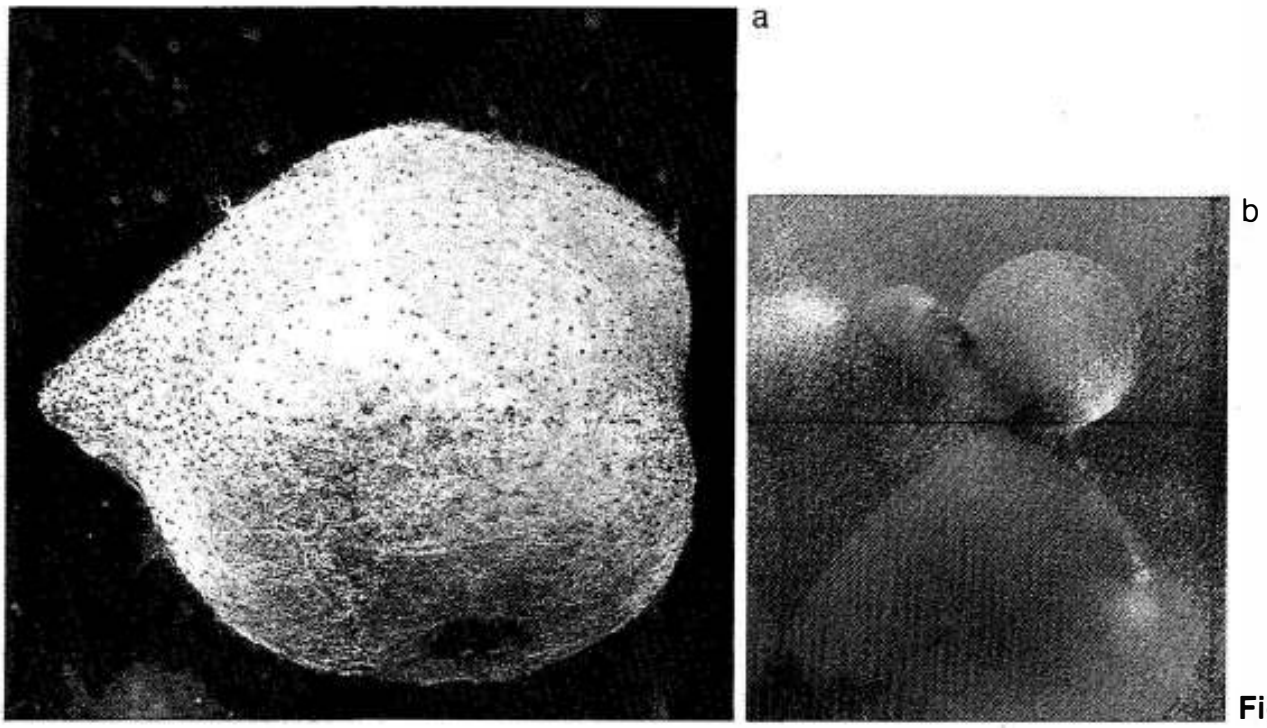


Figure 33A.
(a) Infected chickpea seed after incubation, showing the growth of Phoma medicaginis, *×13, (b) conidial ooze of the fungus, ×113.*

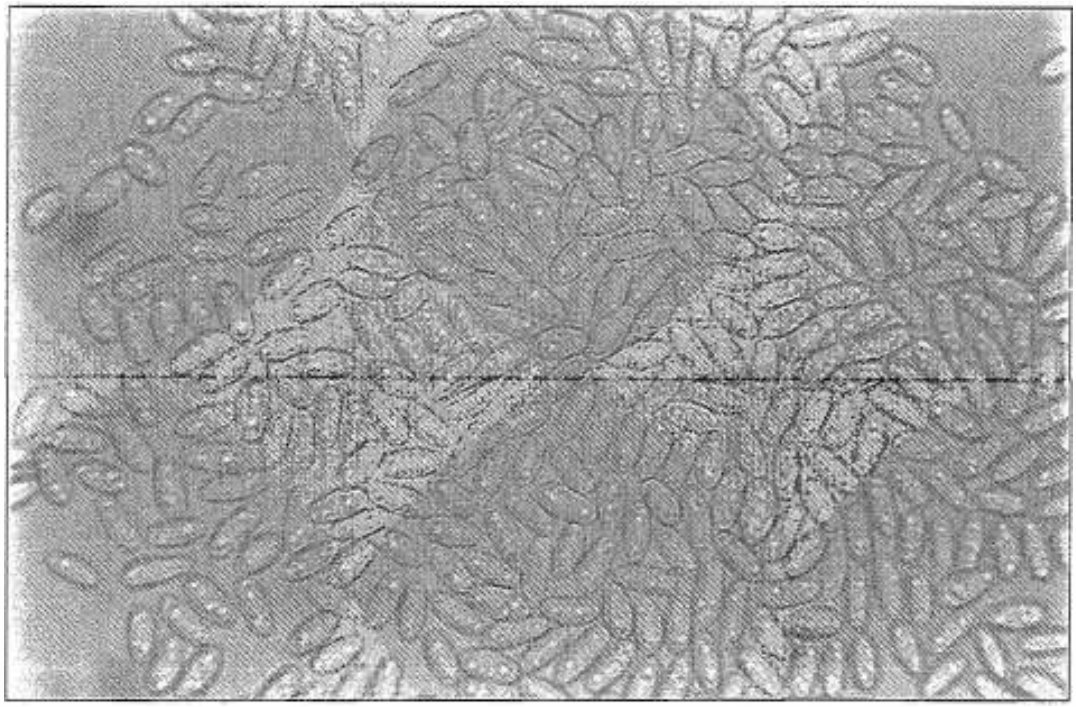


Figure 33B.
Conidia of P. medicaginis, *×1130.*

Pigeonpea

Alternaria alternata (Fr.) Keissler

Alternaria tenuis C.G.Nees

Alternaria alternata causes leaf spot/blight diseases of chickpea, pigeonpea, and groundnut. Infected seeds of pigeonpea and groundnut can be detected after incubation, but when chickpea seeds are heavily infected they can be identified even without incubation. Nonpathogenic forms of this species are often recorded on seeds during incubation tests. Under the microscope, they are identical to the pathogenic forms. So, proof of pathogenicity is required to distinguish them. Dry, infected seeds of chickpea are discolored and shriveled. On incubated seed, the fungus produces woolly or powdery chains of dark brown conidia of variable lengths and shapes (Fig. 34A). The color of the colony is usually olive green to dark brown, but extremely variable. The **mycelium** may be either sparse or abundant and very variable in color, usually light olive green to brown. Hyphae are dark brown, thick, septate, and branched. **Conidiophores** are simple, erect, 40–50 µm long, 2–6 µm thick, and often clustered. They produce darkly pigmented conidia in an acropetal succession of simple or branched chains (Fig. 34B). The chains normally branch at the beak of a spore, or sometimes from the short lateral projection of the beak. **Conidia** have transverse and oblique septa, measure 10–18 × 20–65 µm, and are ovoid to obovoid, obclavate, obpyriform, ellipsoidal, muriform, with an elongated terminal cell. Conidia often have a short conical or cylindrical beak which may be up to one third the length of the conidium, and measure 2–5 × 10–20 µm. (Fig. 34C) Surface walls are either smooth or verrucose and pale to mid-golden brown (Haware et al. 1986; Nene and Reddy 1987; Reddy et al. 1990).

Control. Seed treatment with thiram at about 4 g kg^{-1} satisfactorily controls the seedborne infection in chickpea (Suhag 1973). For pigeonpea, captan or thiram at about 3 g kg^{-1} as slurry treatment or a (1:1) mixture of 30% benomyl + 30% thiram (Benlate T® at about 3 g kg^{-1}) are advised to eradicate the fungus from the seed (Kannaiyan et al. 1980).

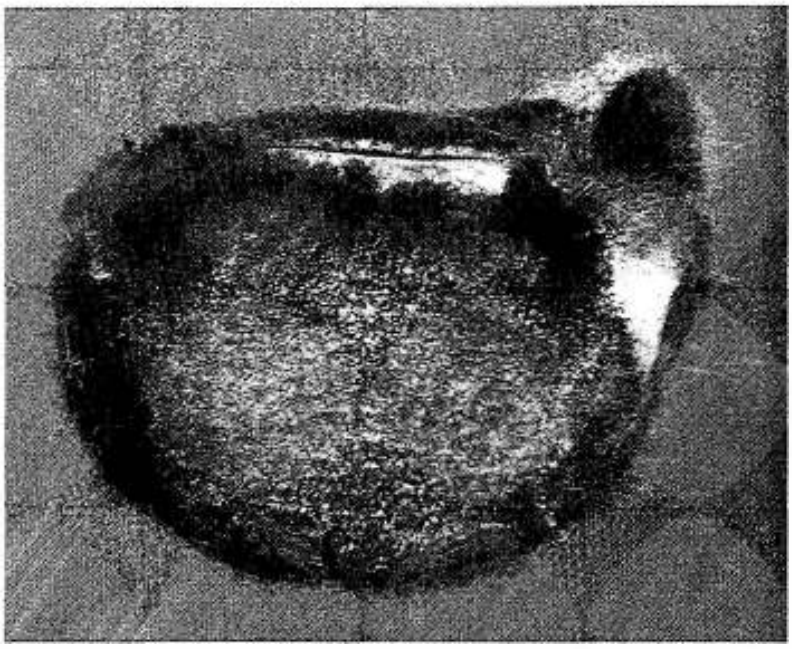


Figure 34A.
Infected pigeonpea seed after incubation, showing the growth of Alternaria alternata, *×18.*

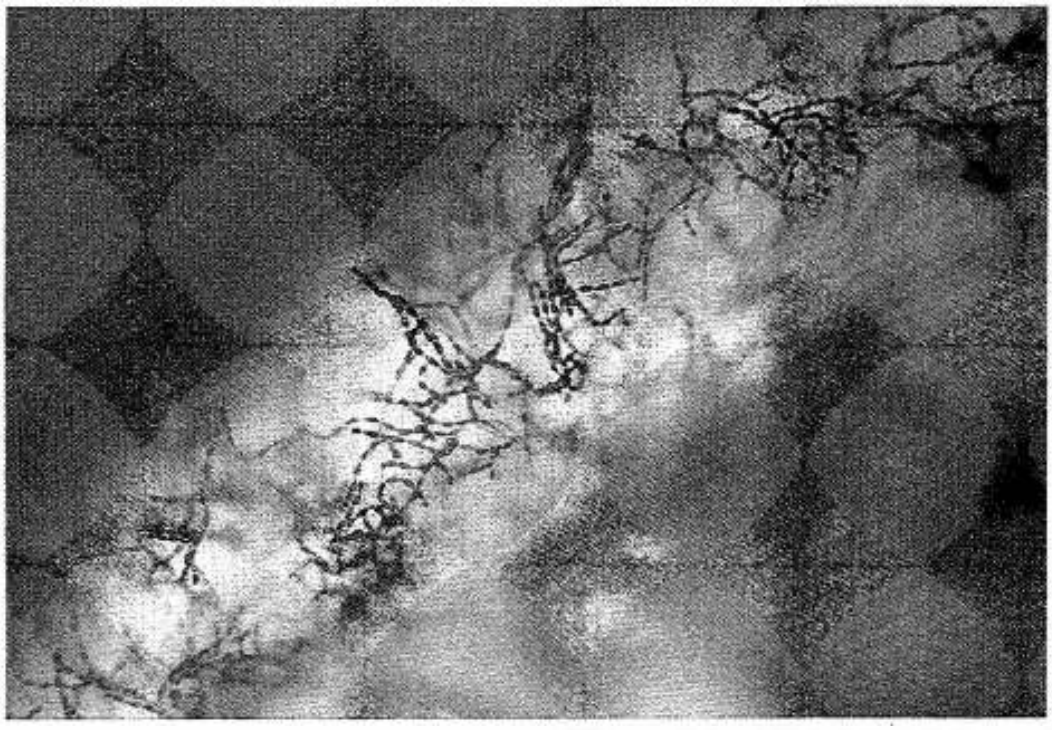


Figure 34B.
Chains of conidia produced by A. alternata, *×113.*

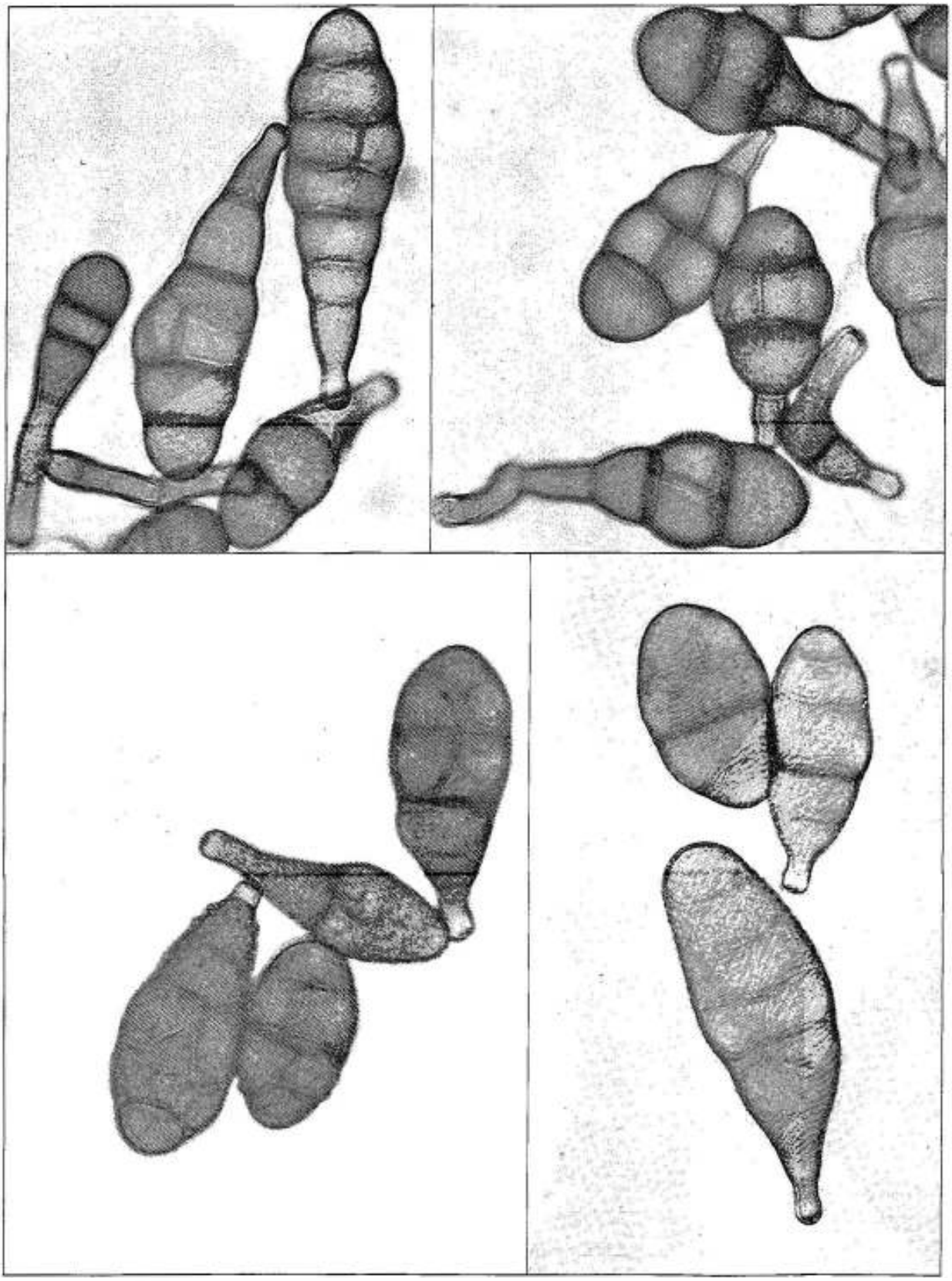


Figure 34C.
Conidia of A. alternata, *×1130.*

Botryodiplodia theobromae Patouillard

Lasiodiplodia theobromae (pat.) Griffin & Maubl.

Botryodiplodia spp and *B. theobromae* cause blight in pigeonpea and collar rot in groundnut. Infected seeds can be detected by visual examination and incubation tests. Dry, infected seeds are usually discolored, shriveled, undersized, and moldy. Sometimes black pycnidia can be seen as pinhead-sized dots during visual examination (Figs. 35Aa, b, and c). The profuse growth of the fungus on incubated seed often results in the seeds' failure to germinate. Dark brown to black pycnidia grow below the seed coat, and can rupture it, resulting in a warty or charred appearance. Pycnidia mature slowly and ooze conidia in a white to dull white or black gelatinous mass. The fungus can be identified under a stereobinocular microscope by the presence of characteristic white and black, long, rough cirrhi (Figs. 35Ba and b). The **mycelium** is superficial, loose, cottony grayish brown to chocolate brown, and may cover the entire seed. **Pycnidia** are mostly immersed, but may be erumpent or superficial. They are separate or aggregated, confluent, globose, black, and 3–5 mm in diameter. Dark brown superficial hyphae can often be seen on their surface. There is no ostiole and pycnidia dehisce irregularly. **Conidia** are of two types: the first are immature, hyaline, single-celled (Fig. 35Ca), and ooze from pycnidia in a white to dull white cirrhus; the second are dark, bicelled, mature, possess longitudinal striations, and ooze from black cirrhi (Fig. 35Cb). Conidia are globose, subovoid to ellipsoid-oblong, thick-walled with a truncate base, and measure 20–30 × 10–15 μm (Gilman 1965; Punithalingam 1976; Neergaard 1979).

Control. Crop health, seed vigor, and optimum storage conditions should be considered while selecting seed lots for export. Discard seeds showing visible signs of infection. Fungicides such as phenyl mercury acetate (Ceresan® at about 2 g kg^{-1}) or dexonal can be used at about 2 g kg^{-1} to eradicate seedborne infection (Kocaturk and Maden 1977).

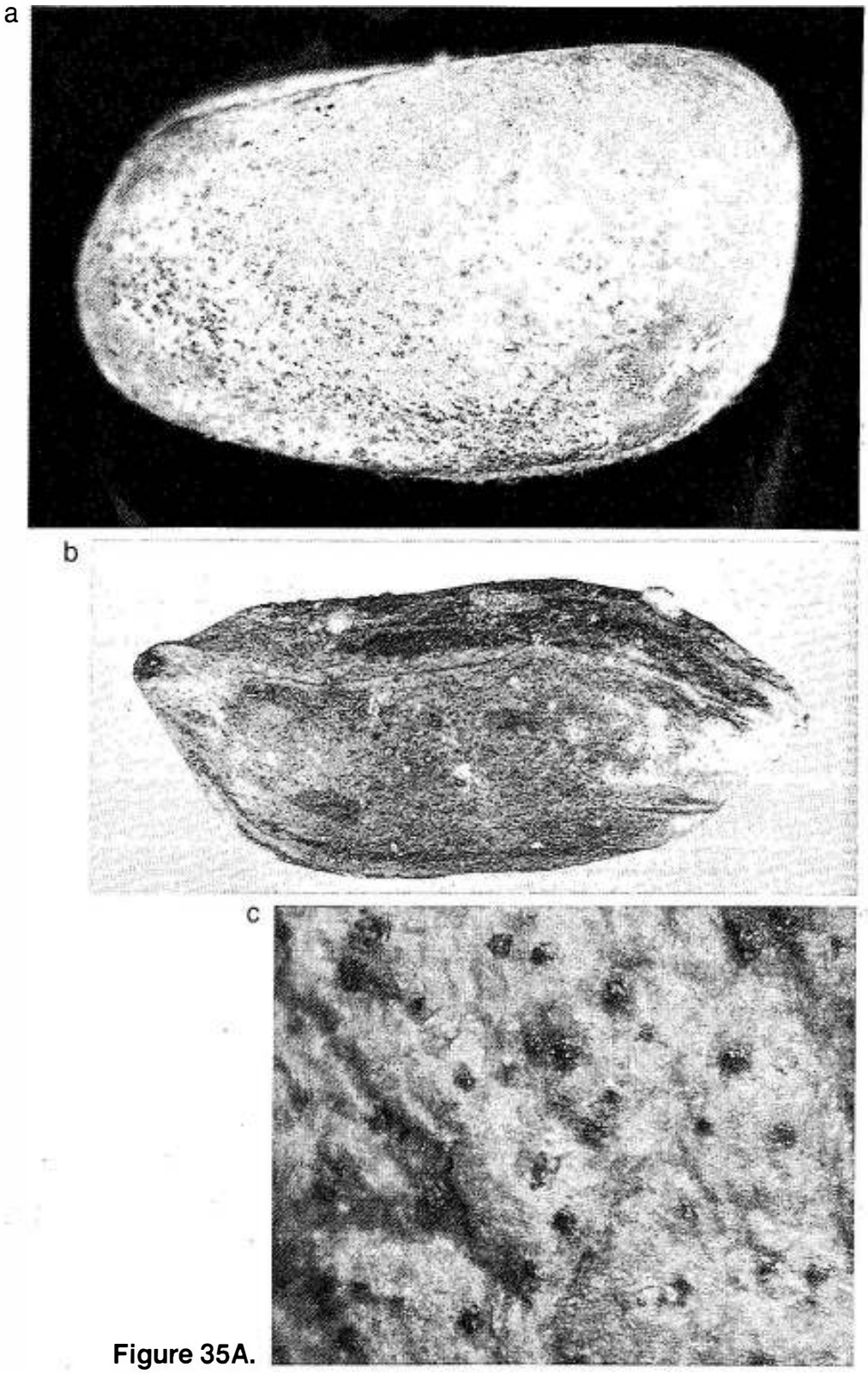


Figure 35A.
Dry seeds of: (a) pigeonpea, ×27, (b) groundnut, ×7, showing the damage caused by Botryodiplodia theobromae, *and (c) pycnidia of the fungus on groundnut seed, ×61.*

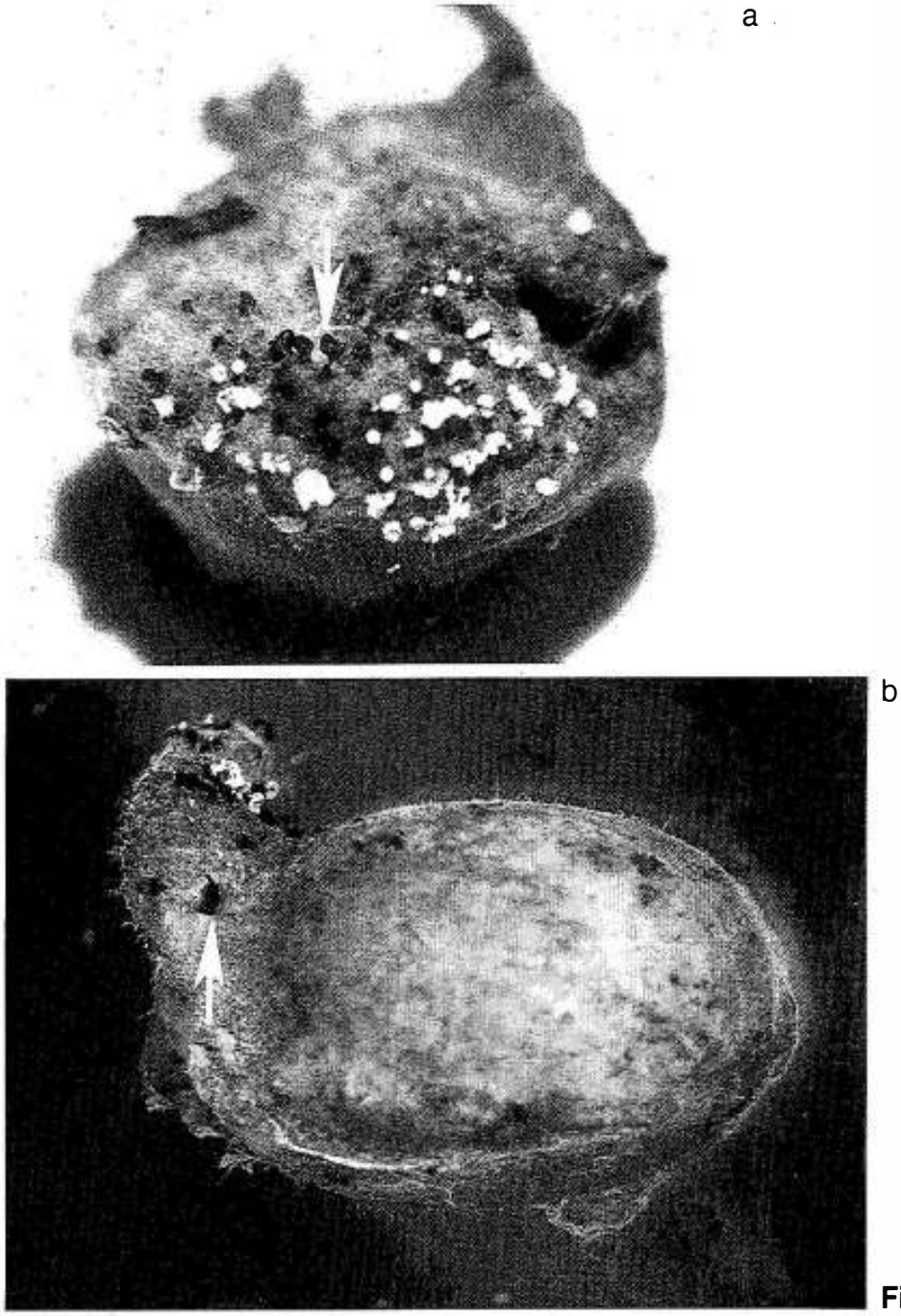
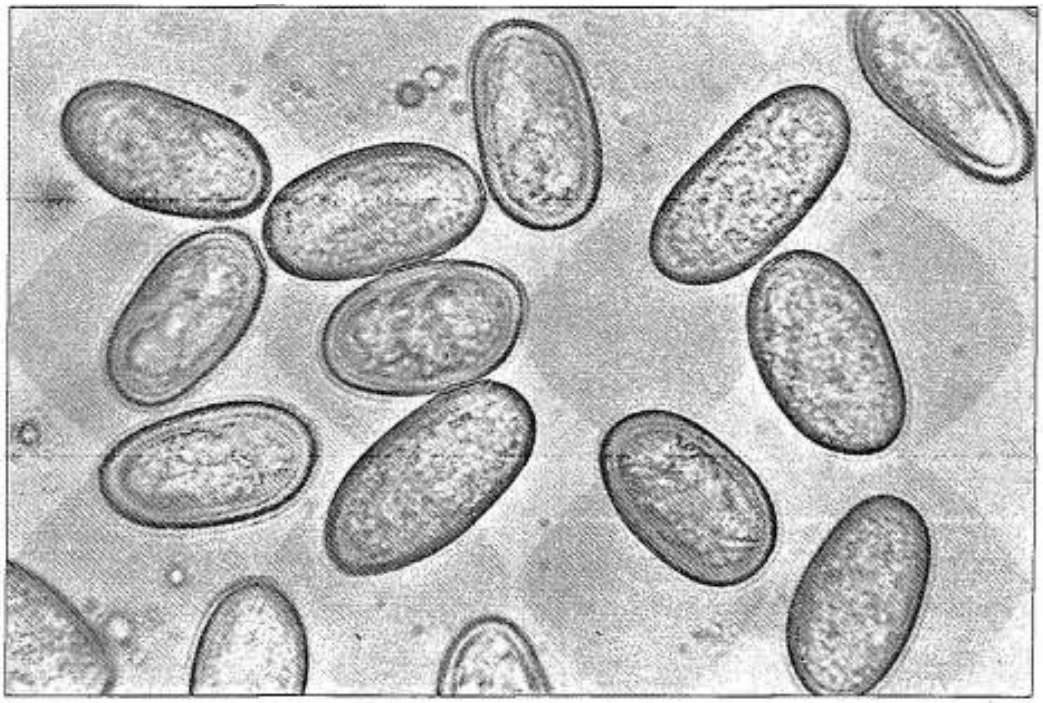


Figure 35B.
(a) Pigeonpea, ×22, (b) groundnut seeds, ×16, after incubation, showing the growth of the fungus consisting of white and black cirrhi (arrowed).

a



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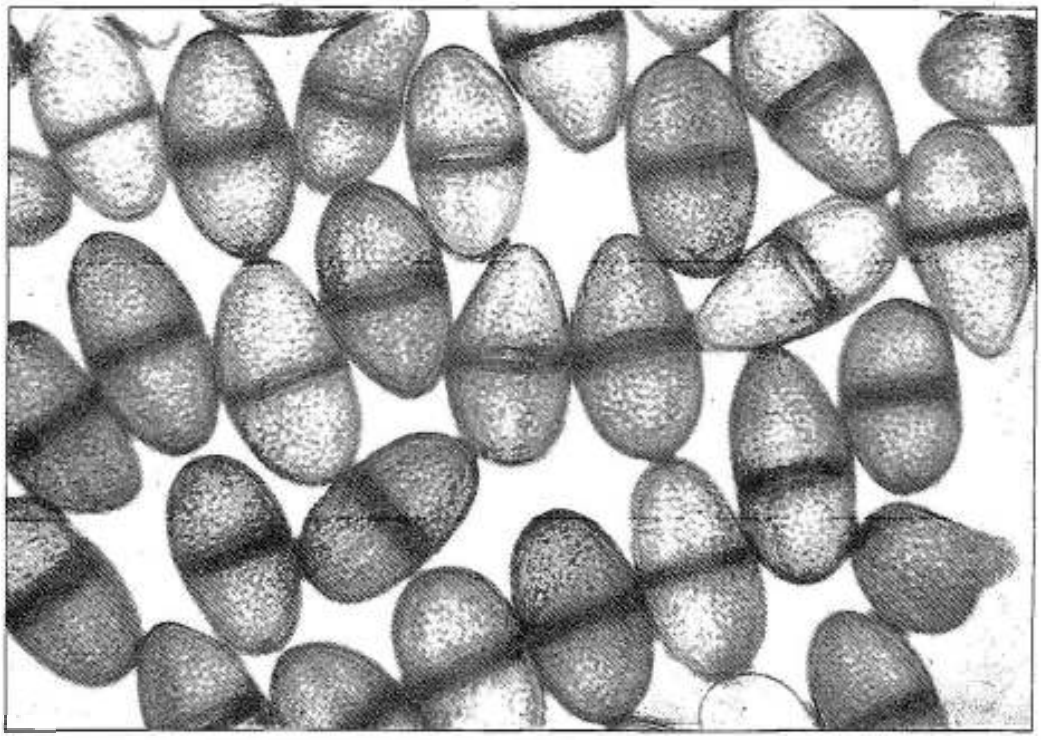

Figure 35C.
Conidia of B. theobromae*: (a) hyaline and single-celled (immature), ×1130, (b) colored and bicelled (mature), ×1130.*

Fusarium udum Butler

This fungus is seedborne, host-specific, and causes vascular wilt of pigeonpea. Infected seeds can be detected by incubation tests (Figs. 36Aa and b). The fungus grows slowly on incubated seed, and produces **mycelium** and conidia, which are formed from simple or verticillately branched conidiophores. These **conidiophores** bear branched monophialides, on the ultimate tips of which conidia are produced. Under a stereobinocular microscope they appear as dry, white, powdery (false) heads (Fig.36B). The philalides (microconidiophores) are subcylindrical to almost doliiform, with a distinct collarette, and measure 8–15 × 2.5–3.5 µm (Fig.36Cb). **Sporodochia** and pionnotes, which are salmon to rose buff colored, are frequently formed. **Microconidia** are single- or bicelled, hyaline, reniform, mostly curved, ovoid to fusoid, scattered (Fig.36Ca). Sporodochia contain masses of very variable **macroconidia** which are hyaline, thin-walled, falcate with a distinct foot cell, and an apical cell that decreases in width towards the tip. Macroconidia are produced on macroconidiophores (Fig.36Cc). The most distinguishing characteristic of the macroconidia are their strongly curved or hooked apices (Fig.36Cd). They are predominantly 3-septate, less frequently 4–5 septate, very rarely 7-septate, and measure 13–50 × 2–4 µm. **Chlamydospores** are usually not produced on the seed but when present, they are intercalary. The walls of the chlamydospores are smooth (Fig.36Ce) (Butler 1906; Booth 1978a; Subramanian 1971).

Control. Avoid collecting seeds from infected plants and fields where the fungus is known to be endemic. Treatment of seed with a (1:1) mixture of benomyl + thiram in a commercial formulation (1.25 g benomyl + 1.25 g thiram kg^{-1}) is advised to eradicate the fungus (Haware and Kannaiyan 1992).

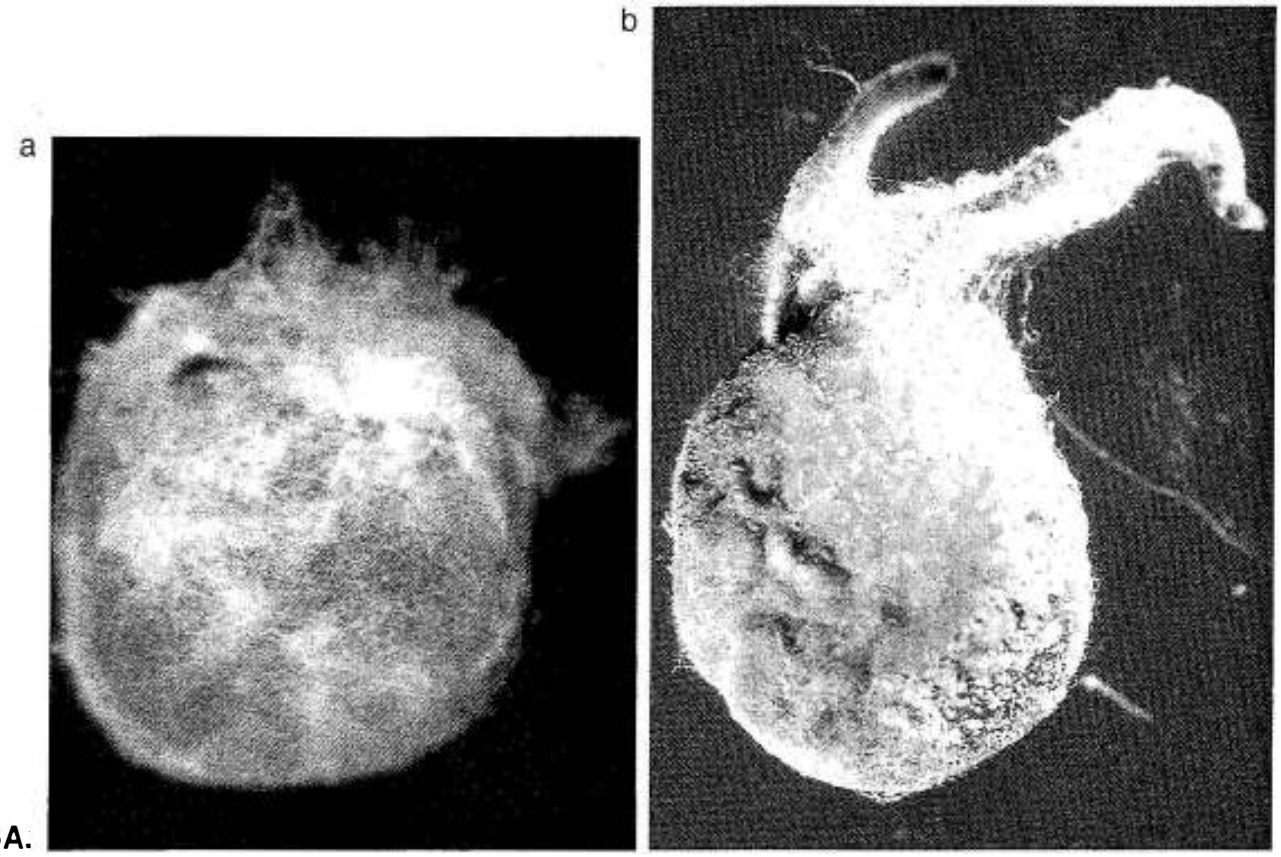

Figure 36A.
Infected pigeonpea seeds after incubation, showing two types of growth of Fusarium udum*: (a) mycelial, ×15, and (b) sporodochial, ×15.*

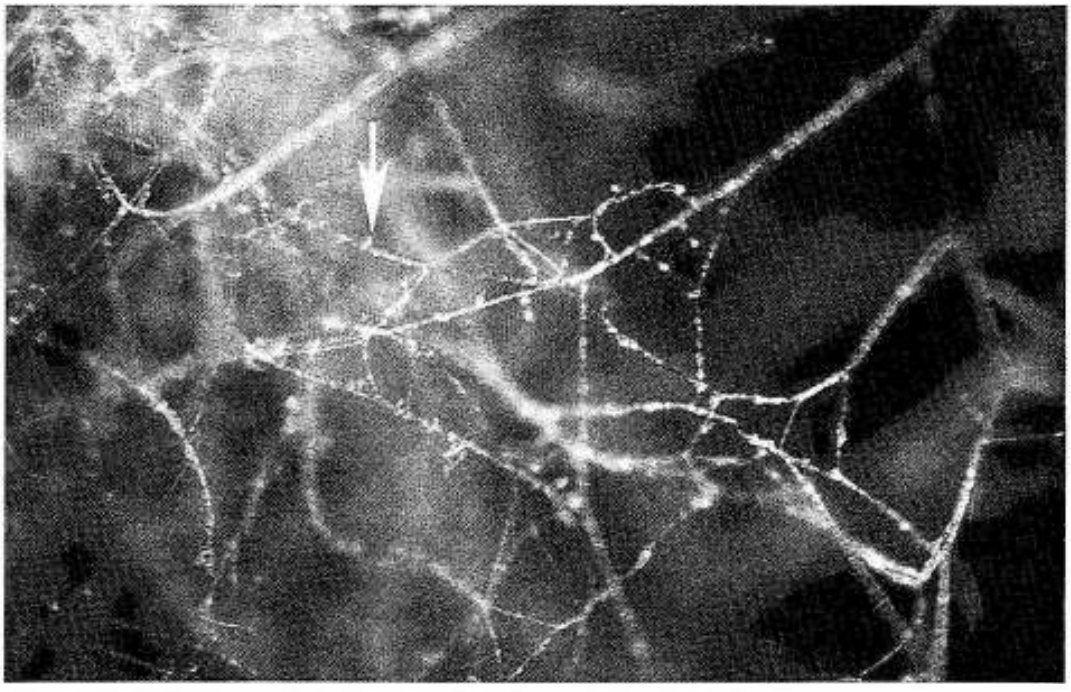

Figure 36B.
False heads (arrowed) produced on the mycelium by F. udum, *×113.*

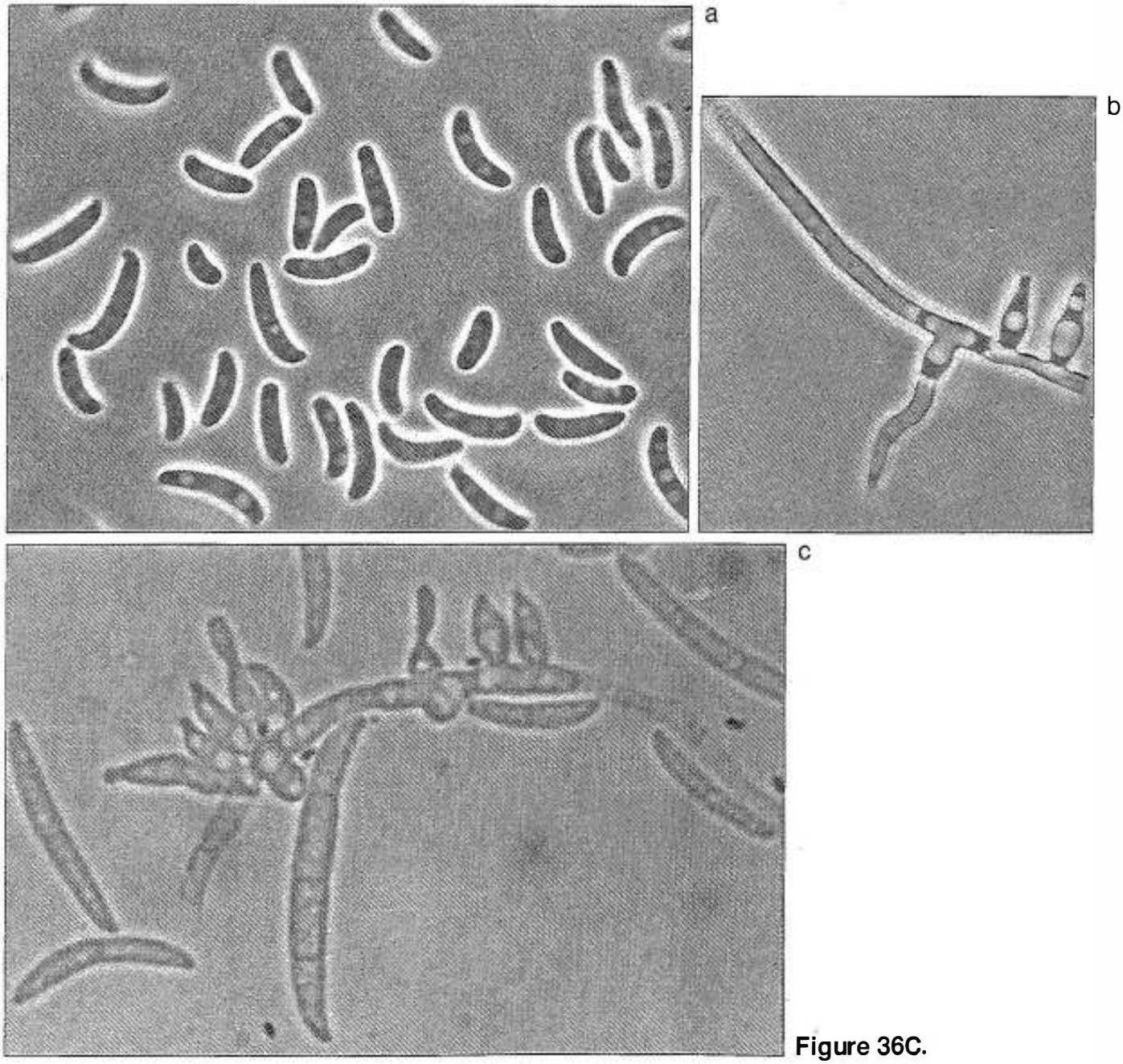

Figure 36C.
(a) Microconidia, ×1130, (b) microconidiophores, ×1130, (c) macroconidiophores, ×1130,

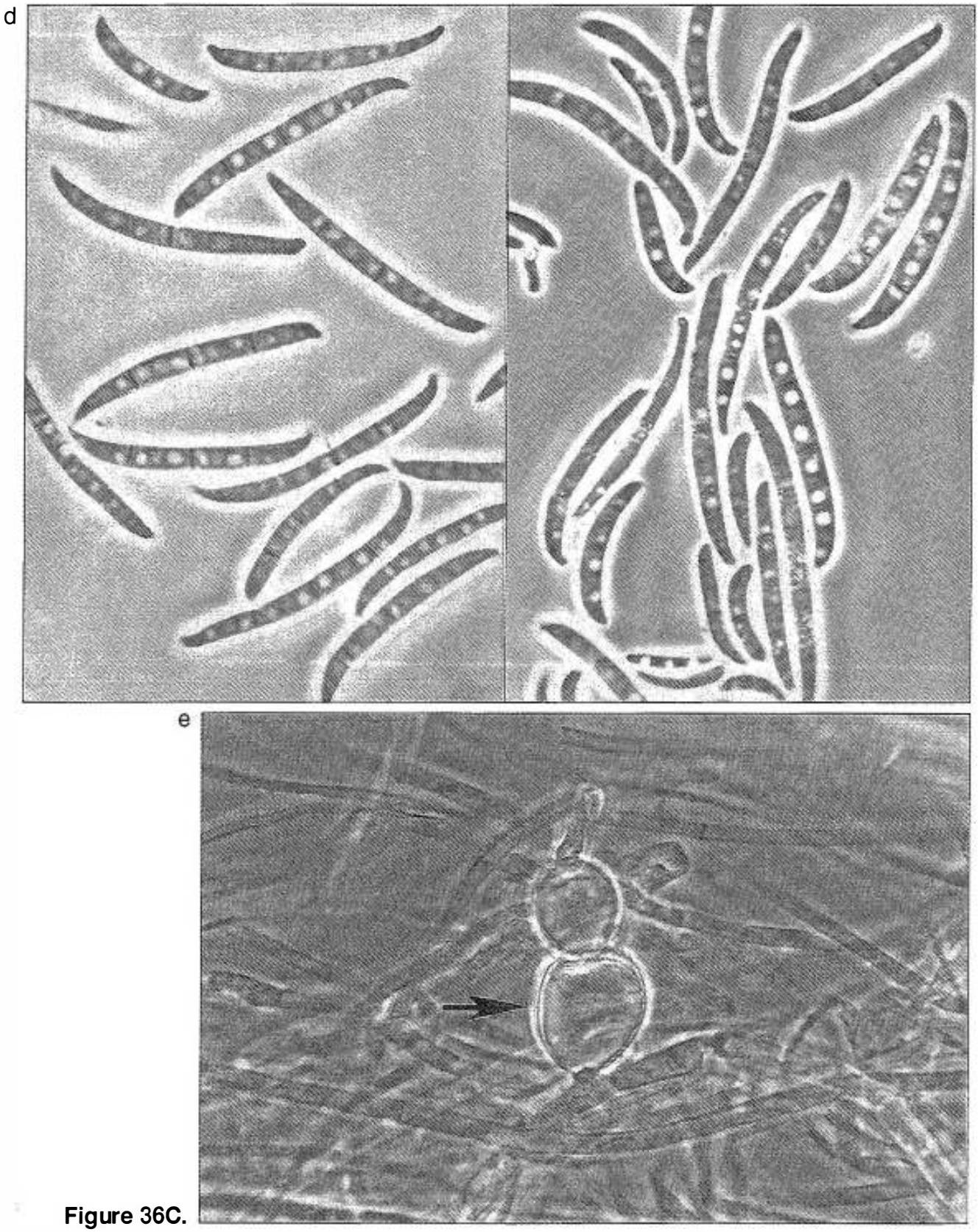

Figure 36C.
(d) macroconidia, ×1130, and (e) chlamydospores (arrowed) of F. udum, *×1130.*

Phyllosticta cajani Rangel

Phyllosticta cajani is known to be seedborne, causing leaf spot of pigeonpea. Infected seeds show visible symptoms after incubation, when the fungus produces darkly pigmented pycnidia, chlamydospores, and mycelium (Figs. 37Aa and b). Sometimes pycnidia are also produced on blotters (during the blotter test) around profusely infected seed. **Pycnidia** may be erumpent or embedded, scattered singly or aggregated into a mass, and are 45–130 µm in diameter (Fig. 37B). Individual pycnidia are globose to subglobose, pyriform or tympaniform, ostiolate with very short beaks, and are rough-surfaced. Mature pycnidia release hyaline, ovoid to elongate, single-celled conidia in a dirty white to light cream gelatinous mass. The **conidia** are globose, obovoidal, ellipsoidal or clavate, broadly rounded, apically flattened or indented, and measure 2.0–6.0 × 1.5–2.5 µm (Fig. 37Ca). They are surrounded by a slime layer, have an apical appendage and characteristic greenish guttules which distinguish this fungus from the fungi in the genus *Phoma*. **Chlamydospores** are dark brown and resemble the thick-walled conidia of *Alternaria* (Fig. 37Cb). They are irregularly septate into transverse and oblique septa (Kumar 1980; Holliday 1980).

Control. Avoid collecting seeds from endemic fields or infected plants. Information on specific fungicidal seed treatment is not available.

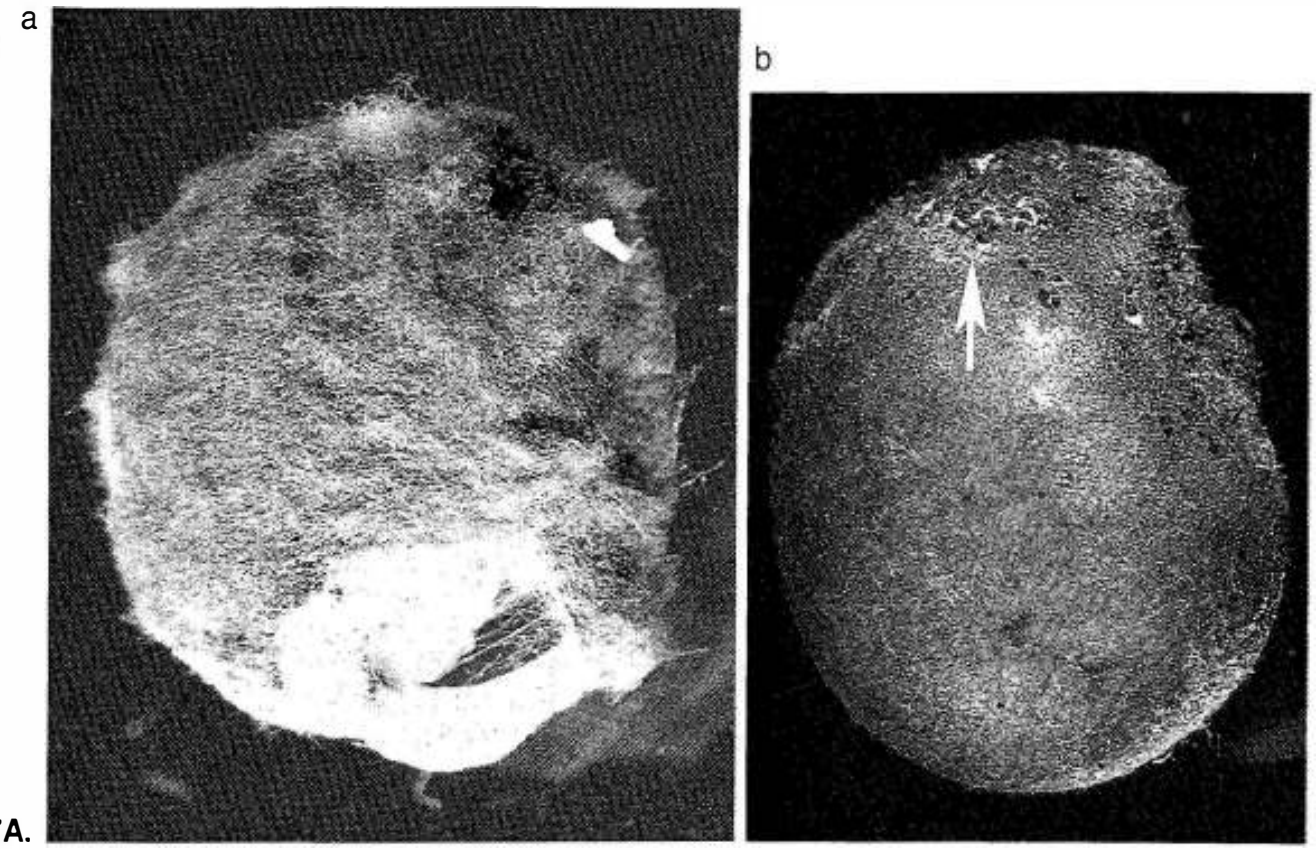

Figure 37A.
Infected pigeonpea seeds after incubation, showing two types of growth of Phyllosticta cajani: *(a) mycelial, and (b) pycnidial (arrowed), ×29.*

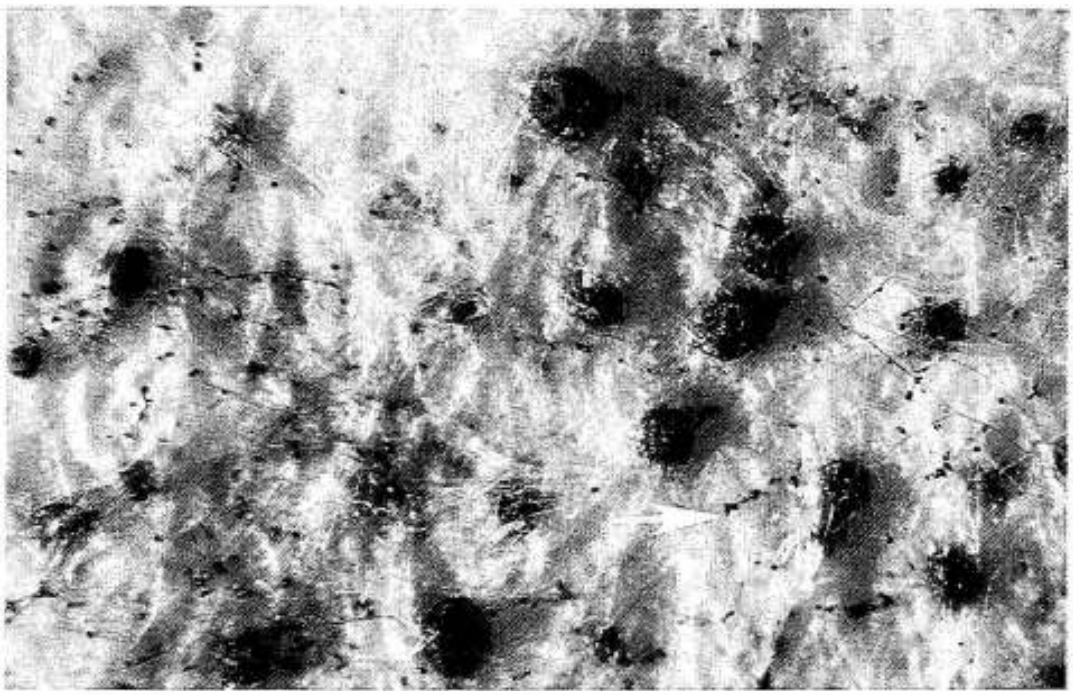

Figure 37B.
Chlamydospores (arrowed) and pycnidia produced by P. cajani, *×335.*

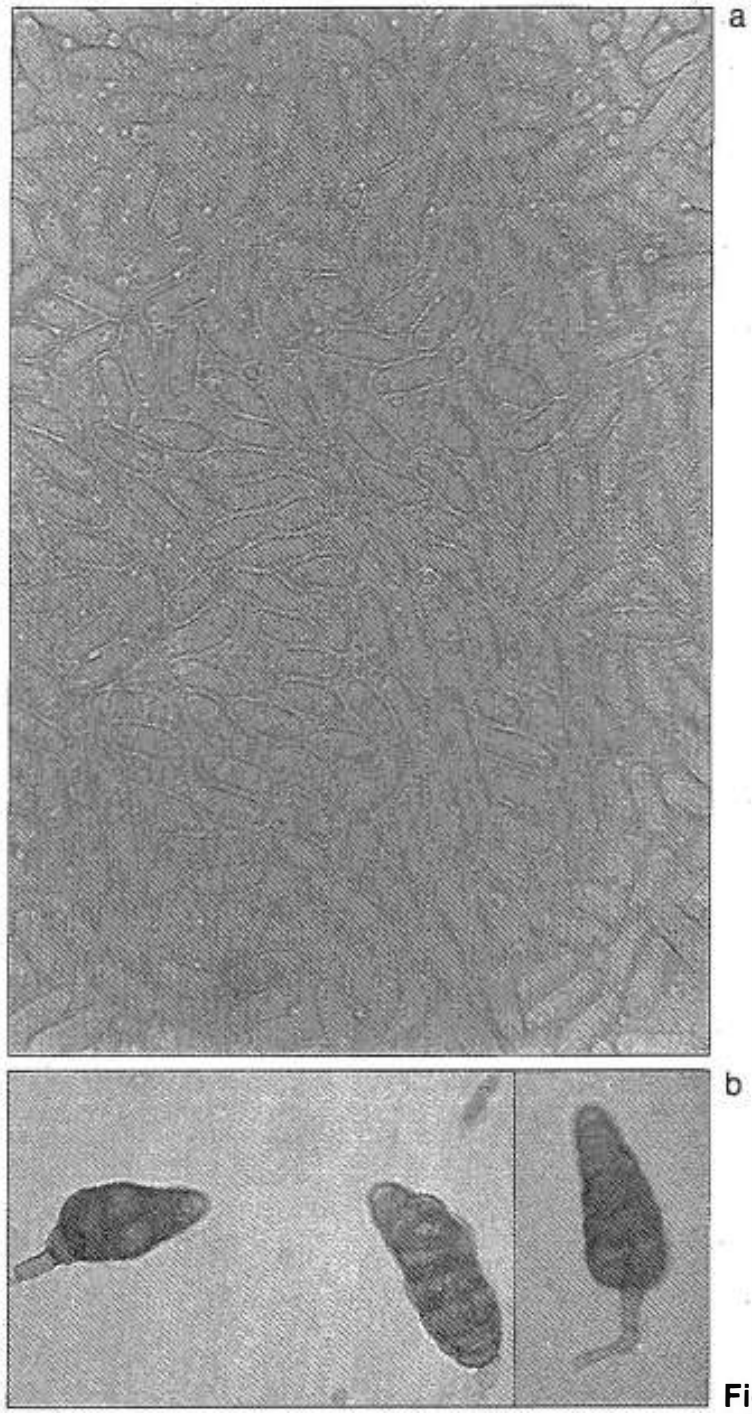

Figure 37C.
(a) Conidia, ×1130, and (b) chlamydospores of P. cajani, *×225.*

Groundnut

Aspergillus flavus Link ex Fr.

The fungus represents a member of an aggregate group of species frequently observed on groundnut seeds. *Aspergillus flavus* is parasitic on the groundnut crop causing aflaroot disease. Infected groundnut seeds can be detected by visual examination and incubation tests. Masses of yellow-green ball-like structures supported on stalks are visible on dry infected seeds under a stereobinocular microscope (Fig. 38Aa and b). Moldy, discolored, shriveled, and damaged seed may also be infected. On incubated seed, the fungus produces compact globose to radiate conidial heads in shades of green (Fig. 38Ba). The **mycelium** is mostly submerged in the seed coat and forms a white to gray, tough felty mass. **Conidiophores** of the fungus stand erect on the seed. They are simple, unbranched, colorless, transparent, smooth, up to 1 mm long, and 10–20 µm thick (Fig. 38Bb). The apex of the conidiophore is inflated into a vesicle upon which radiating phialides are formed (Fig. 38Ca). **Conidial heads** are biseriate, 300–500 µm in diameter, globose to radiate or columnar, very light to deep yellow green, olive brown or brown. **Conidia** are hyaline, single-celled, and produced in chains. They are typically globose to subglobose, 3–6 µm in diameter, sometimes elliptical to pyriform, and conspicuously echinulate (Fig. 38Cb) (Raper et al. 1965; Onions 1966a; Jackson and Bell 1969; Porter et al. 1984).

Control. As a result of contamination by this fungus aflatoxins are produced in seed tissues. These toxins are highly resistant and hazardous to both human and animal consumers. Contamination drastically reduces the value of the crop. So prevention of fungal infection is very important. Groundnuts should be harvested at the right time and damage should be avoided while lifting and handling in the field. Rapid drying of groundnuts immediately after the harvest to less than 9% moisture content prevents infection (Feakin 1973). Avoid moisture damage and insect infestation during storage before export. Discard discolored and moldy kernels during visual examination. Fungicides such as captan, thiram, or carbendazim can be applied at about 3 g kg^{-1} seed to reduce infection (Ghewande et al. 1987).

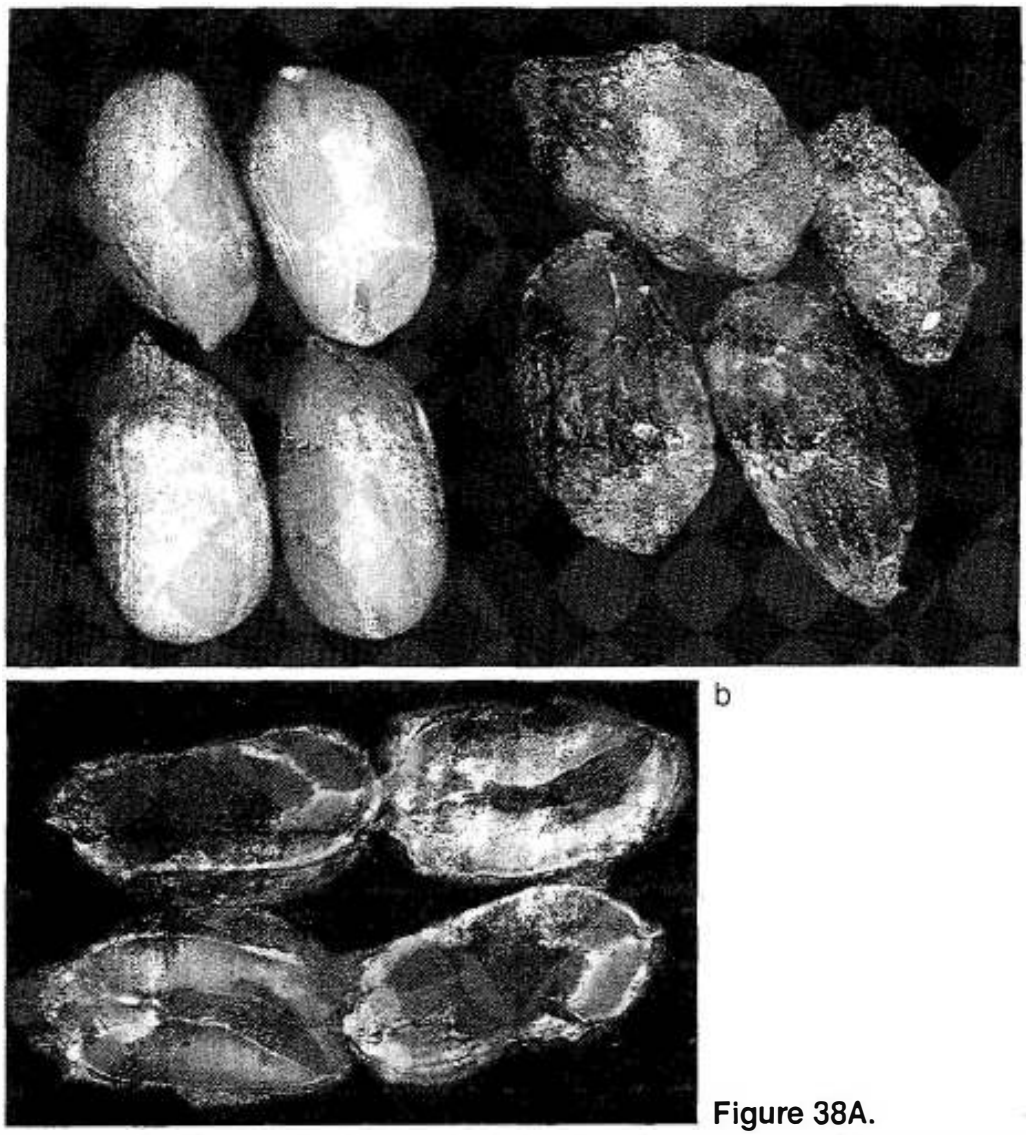

Figure 38A.
(a) Dry seeds of groundnut showing damage caused by Aspergillus flavus *(right), and healthy seed (left), ×7, and (b) internal damage caused by the fungus, ×10.*

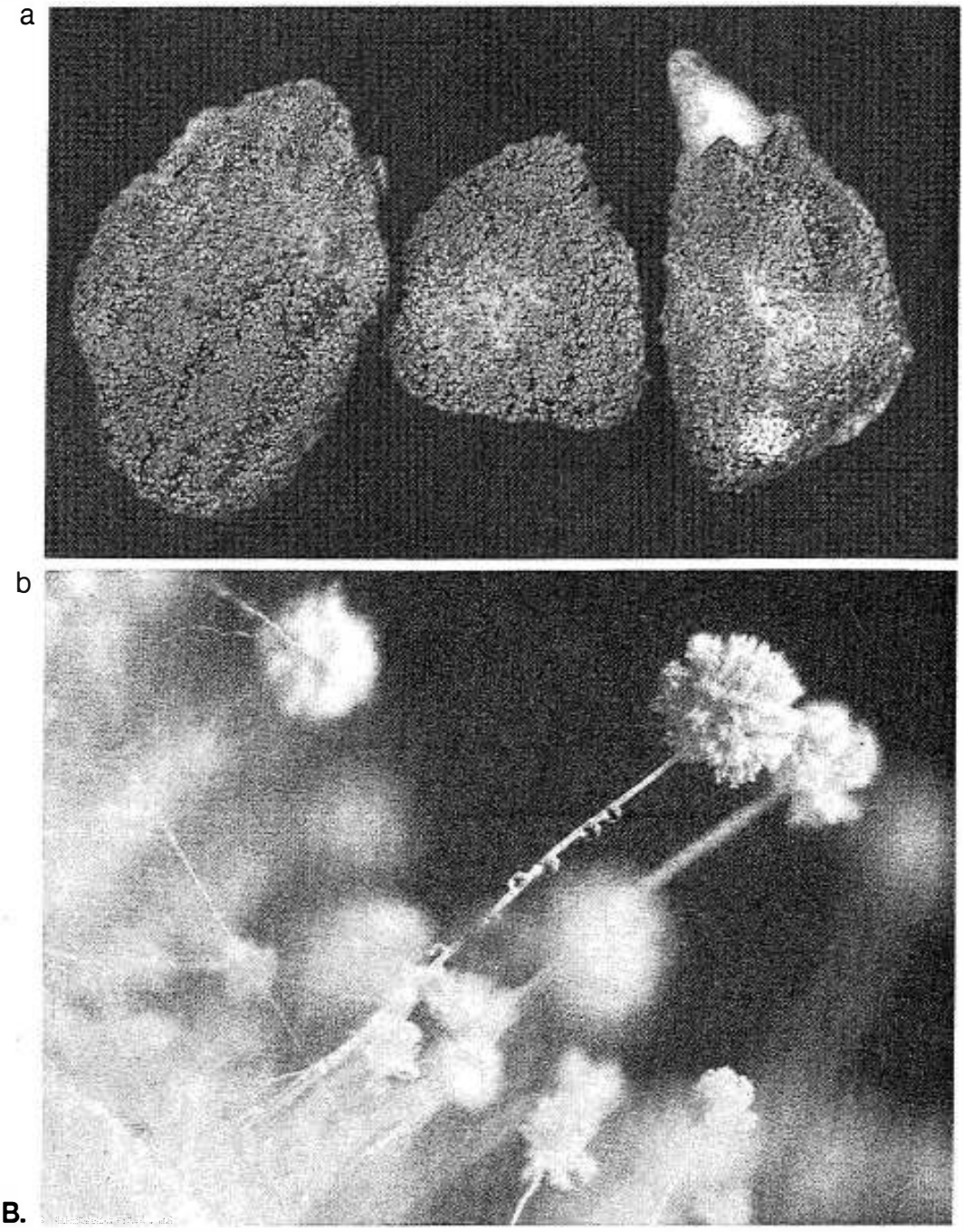

Figure 38B.
(a) Infected groundnut seed after incubation, showing the growth of A. flavus, *×15, and (b) conidiophores with conidial heads, ×113.*

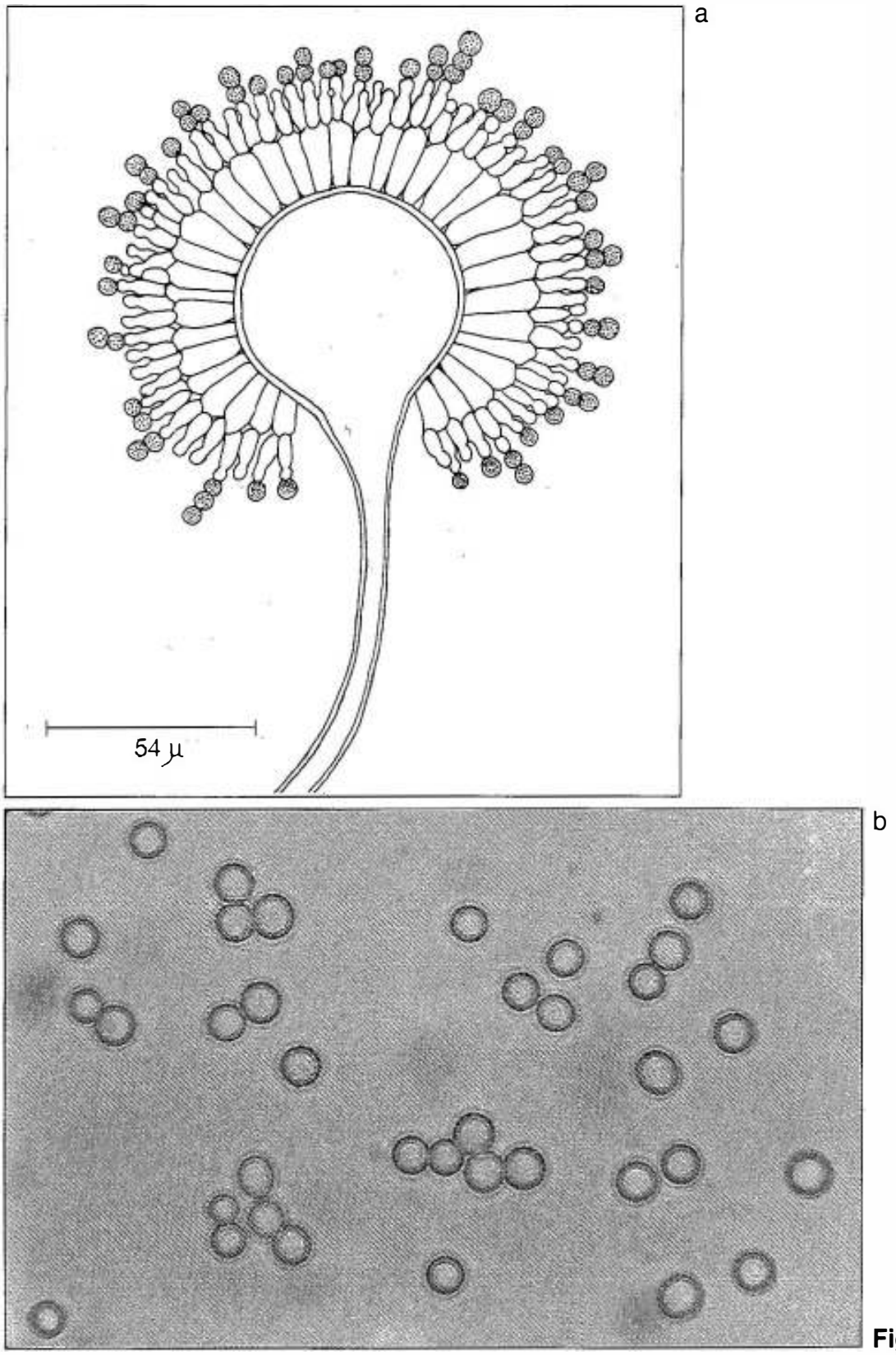

Figure 38C.
(a) Biseriate conidial head, and (b) conidia of A. flavus, *×1130.*

Aspergillus niger van Tieghem

Aspergillus niger is parasitic on groundnut and causes crown rot and collar rot diseases. Seedborne infection can be detected by incubation tests. On incubated seed, the fungus produces abundant conidial heads which are in shades of black (Fig. 39Aa). It also produces sclerotia which are globose to subglobose, and appear as woolly balls on the seed surface (Fig. 39Ab). The **mycelium** is scanty, hyaline to white or light yellow. **Conidiophores** arise directly from the seed coat and are 3 mm long, 15–20 µm in diameter, hyaline to light brown, long, thin, unbranched, erect, brittle, and terminate in an inflated apex upon which radiating phialides are formed. **Conidial heads** initially appear globose, but subsequently split into a few to several irregular or well-defined divergent columns of conidial chains. They are black, globose, or radiate (Fig. 39Ac). **Conidia** are produced in chains on the sterigmata. They are single-celled, pale to dark brown, more or less globose, 4–5 µm in diameter, with low to prominent ridges and echinulations that are usually discontinuous with rough or echinulate surfaces (Fig. 39Ba). Sometimes **sclerotia** are also produced on the seed and appear as bright orange woolly balls. They are globose to subglobose, thick-walled, and hard, containing numerous Hülle cells (Fig. 39Bb) (Raper et al. 1965; Onions 1966b; Porter et al. 1984).

Control. Discard moldy and damaged seed during visual examination. Seed treatment with captan at about 3 g kg^{-1} is advised to reduce the seedborne infection (Ghewande et al. 1987). Seed treatment with carbendazim, benomyl or carboxin at about 2.5 g kg^{-1} is also effective to control the seedborne inoculum (Shekhawat et al. 1986).

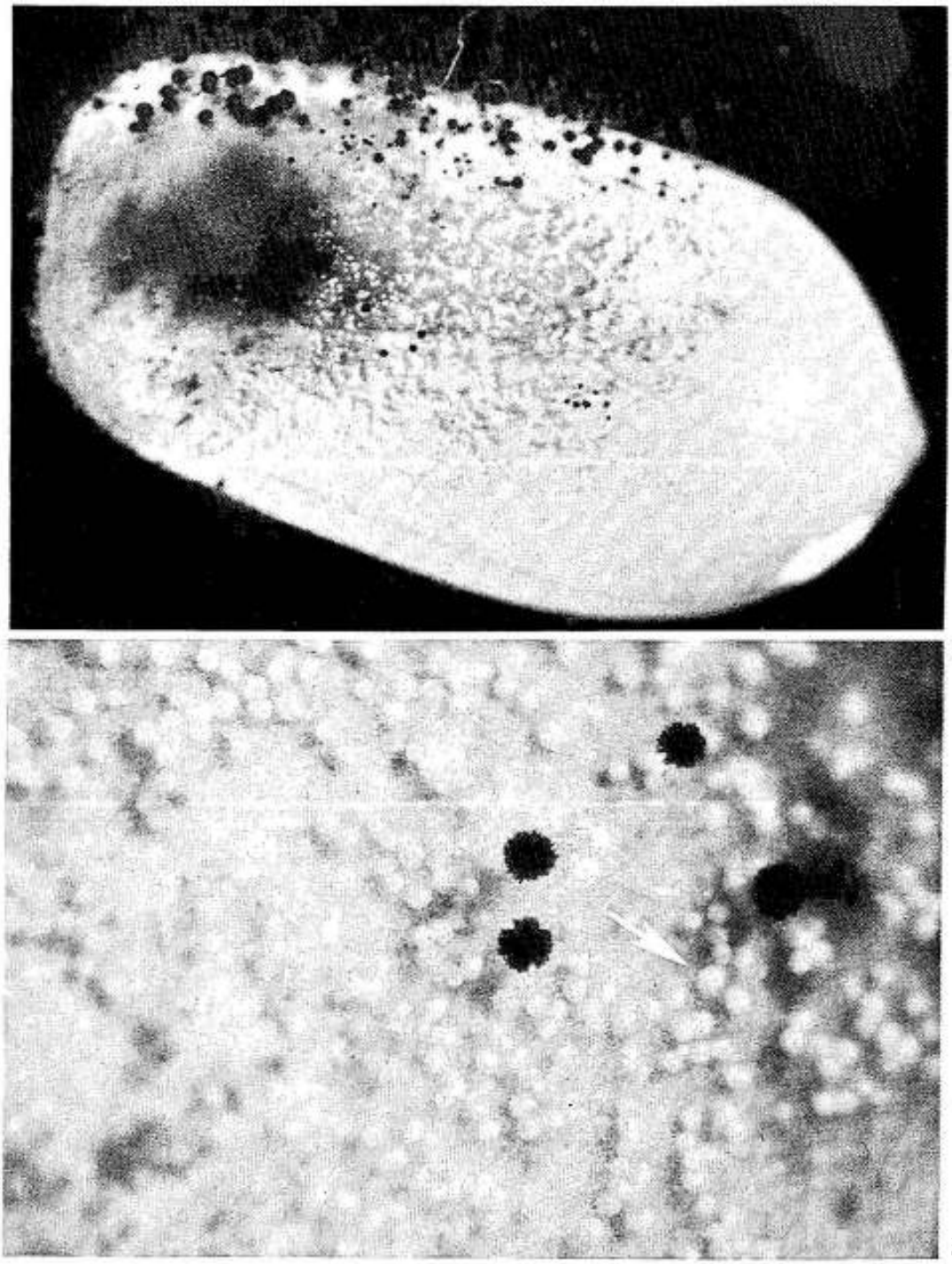

Figure 39A.
Infected groundnut seed after incubation, showing: (a) growth of Aspergillus niger, *×15, and (b) sclerotia (arrowed), ×57, and*

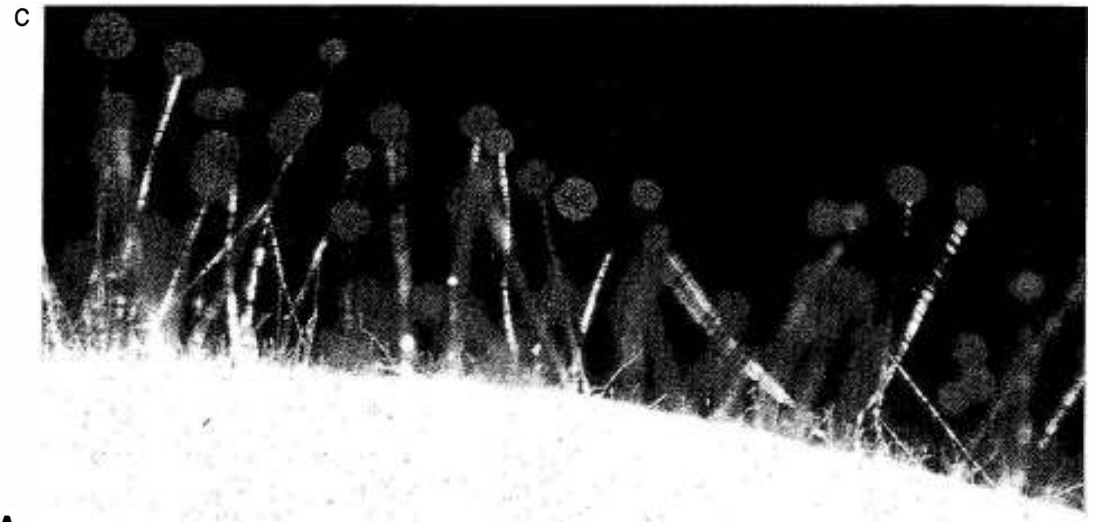

Figure 39A.
(c) conidiophores with globose conidial heads of A. niger, *×57.*

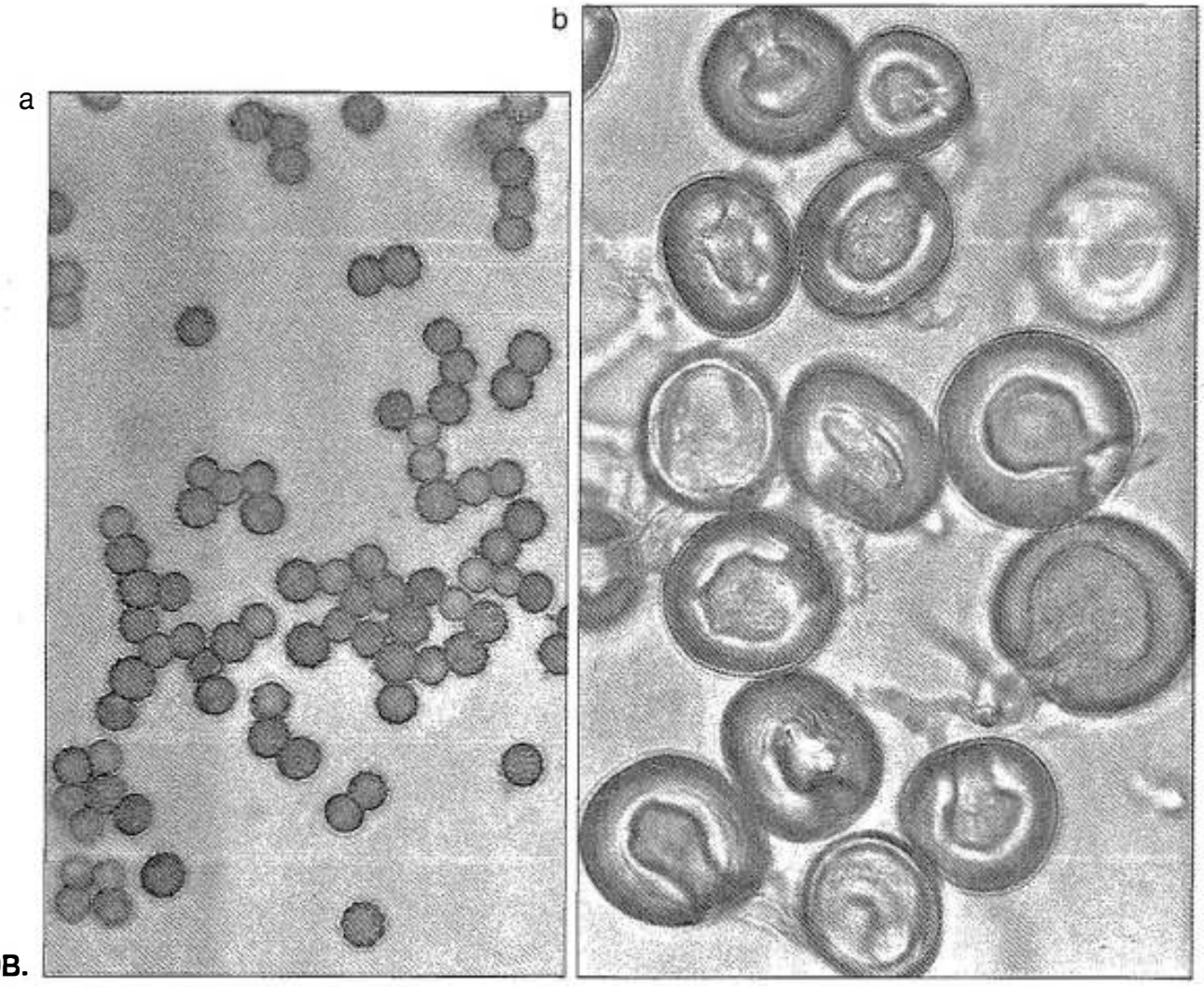

Figure 39B.
(a) Conidia, ×1130, and (b) Hülle cells of A. niger, *×1130.*

Fusarium equiseti (Corda) Sacc.

Fusarium equiseti causes leaf spot of groundnut. Seedborne infection can be detected by incubation tests. Sporodochial forms are frequently produced on incubated seed. Mycelial forms may also occur, but are rare. The growth of the fungus consists of orange sporodochia covering part, or the entire surface of the seed (Fig. 40A). Mycelial forms of this fungus produce few microconidia on the aerial mycelium. These **microconidia** are oval to comma-shaped, produced on microconidiophores, and measure 5–24 × 2–6 µm (Fig. 40Ba). Sometimes the fungus also produces a white to beige, fluffy, compact or loose mycelium in which light orange mucoid pionnotes are present. **Sporodochia** are irregular in size and shape. The sporodochia and pionnotes consist of masses of **macroconidia**, which are hyaline, 3–5 septate with thick septa, 10–87 × 2.3–6.5 µm, and curved; they have a distinct foot-shaped basal cell, while the apical cell is long and extended (Fig. 40Bb). They are produced on branched and unbranched monophialides of the conidiophore (macroconidiophores) (Fig. 40Bc). **Chlamydospores** are produced abundantly on mycelial forms and are 8–20 µm in diameter. They are very prominent, thick, and have roughened walls. Sometimes thick, smooth-walled, globose to subglobose chlamydospores are also produced (Fig. 40Bd) (Booth 1978b; Porter et al. 1984).

Control. Discard moldy, shriveled and damaged seeds during visual examination. Information on fungicidal seed treatment to eradicate the fungus is not available.

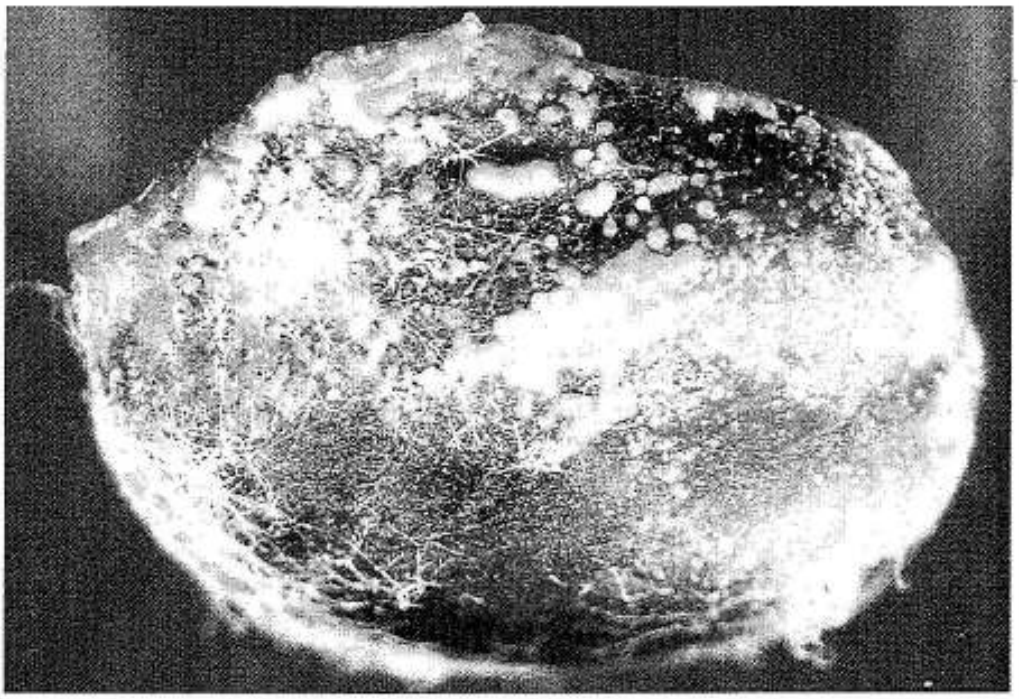

Figure 40A.
Infected groundnut seed after incubation, showing the growth of Fusarium equiseti, *×20.*

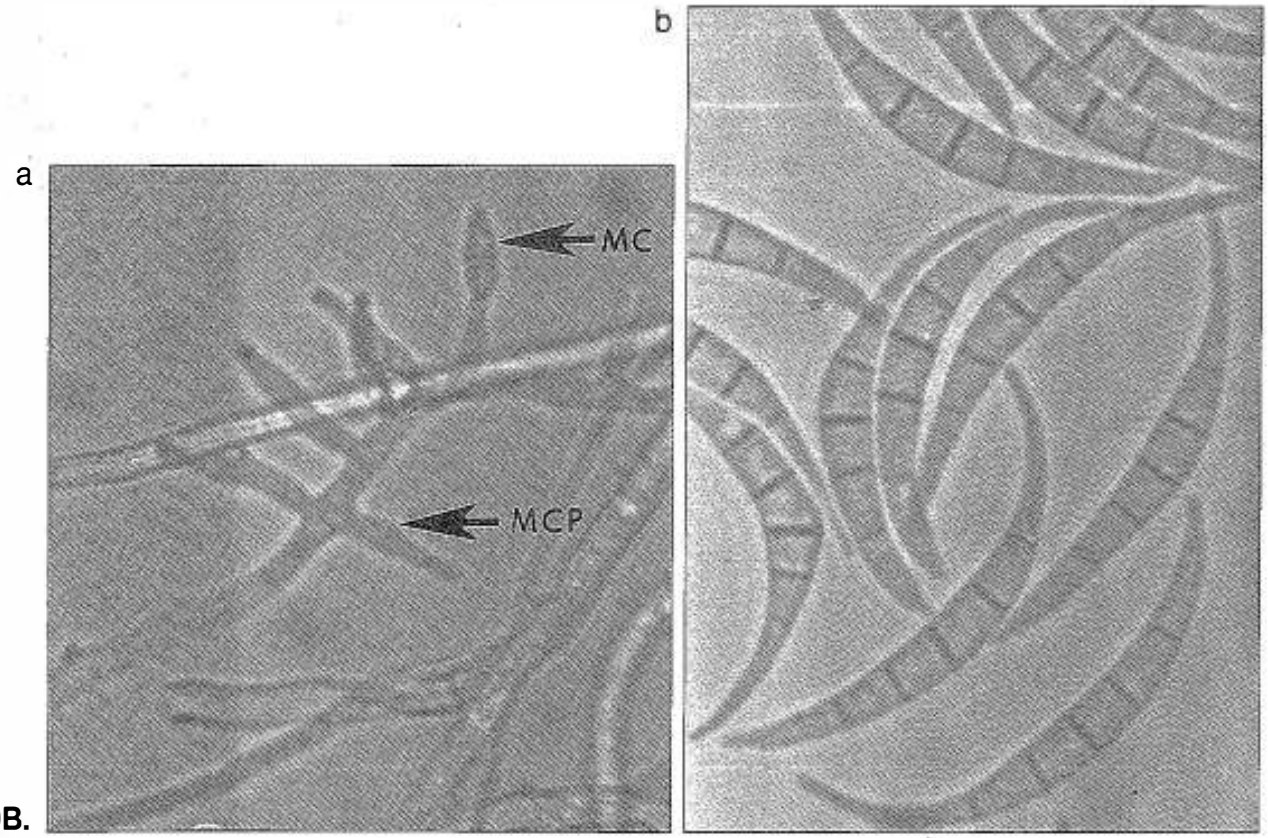

Figure 40B.
(a) Microconidiophores (MCP) and microconidia (MC), ×1130, (b) macroconidia, ×1130,

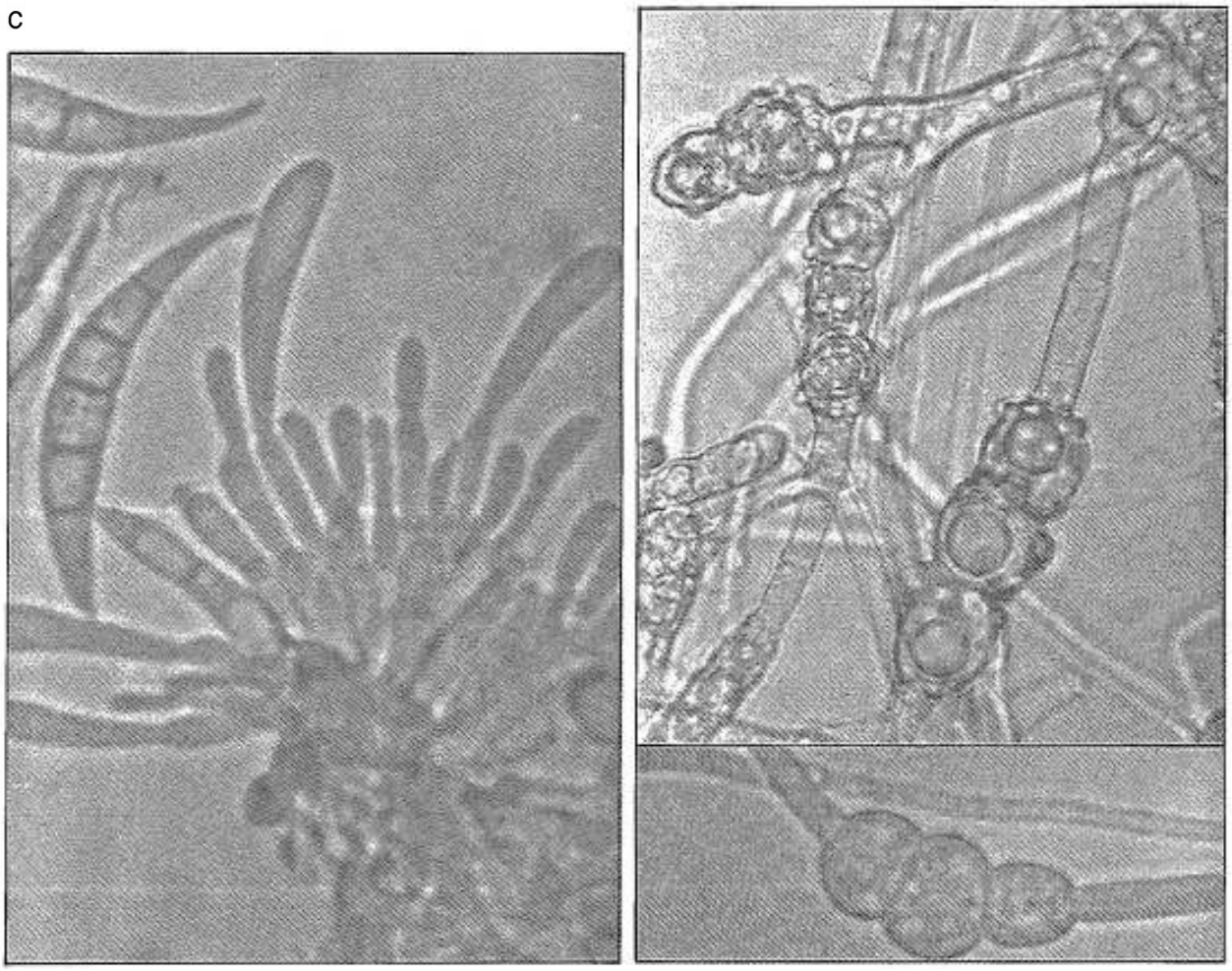

Figure 40B.
(c) macroconidiophores, ×1130, and (d) chlamydospores of F. equiseti, *×1130.*

Fusarium oxysporum Schlechtend emend Sny. & Hans.

Fusarium oxysporum attacks groundnut seedlings and plants, causing damping off and wilts. The pathogen infects groundnut seed through the soil and infected plants. Both the macroscopic and microscopic characteristics of this fungus are very variable. Infected seeds can be detected by visual examination and incubation tests. When the damage is profuse, dry seeds of groundnut emit a golden yellow florescence under UV-light (Fig.41A). On incubated seed, the fungal growth consists of mycelium, sporodochia, and spores (Fig. 41B). Usually sporodochia are not produced on the seed but when produced, they are completely covered by the white to light pink aerial mycelium of the fungus. Individual hyphae produce 8–12 × 2.5–30 μm, unbranched and branched monophialides. These phialides are very short and bear microconidia on false heads (Fig.41C). The **microconidia** produced on microconidiophores, are abundant, hyaline, single-celled, oval, or elliptical, and measure 4–13 × 2–3.5 μm (Fig.41Da). Macroconidia are not abundant, and are mostly produced on the pale orange sporodochia. **Macroconidia** produced on macroconidiophores are hyaline, mostly 3-septate, sometimes up to 5-septate, measure 18–56 × 2.7–6 μm, falcate to almost straight, thin-walled, with a curved apical cell and slightly foot-shaped basal cell (Fig.41Db and c). **Chlamydospores** are terminal and intercalary, irregular in shape, thick-walled with smooth surfaces, and measure 6–11 μm in diameter (Fig.41Dd) (Booth 1970; Porter et al. 1984; Kolte 1984).

Control. Discard moldy and shriveled seed during visual examination. Seed dressing with fungicides such as carbendazim at about 2 g kg^{-1} is advised to reduce the seedborne inoculum (Ghewande et al. 1987).

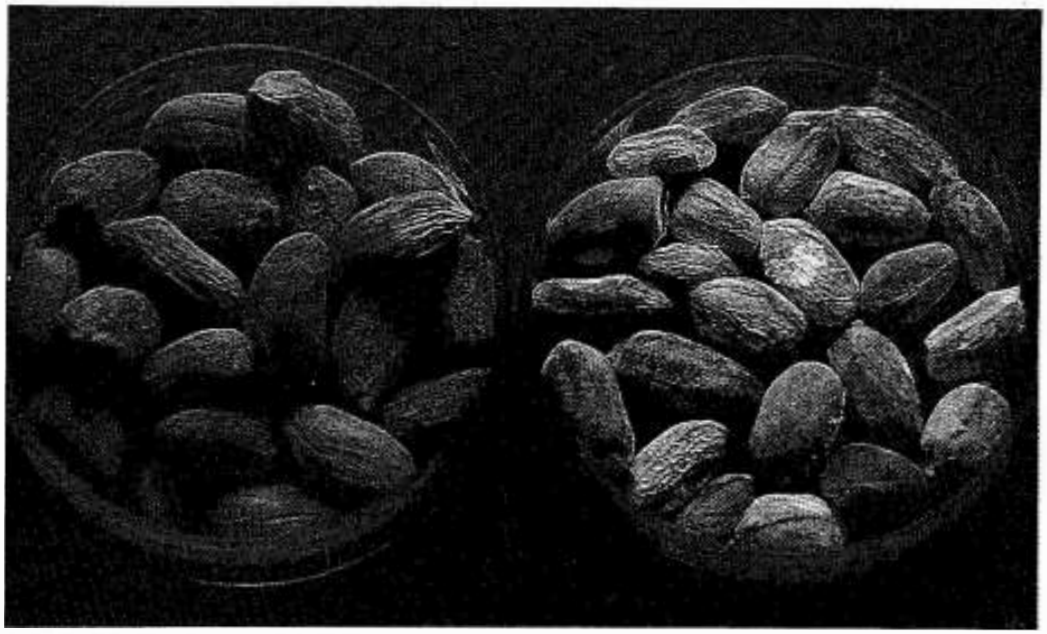

Figure 41A.
UV-light photograph of dry groundnut seeds damaged by Fusarium oxysporum *(right), and healthy seeds (left).*

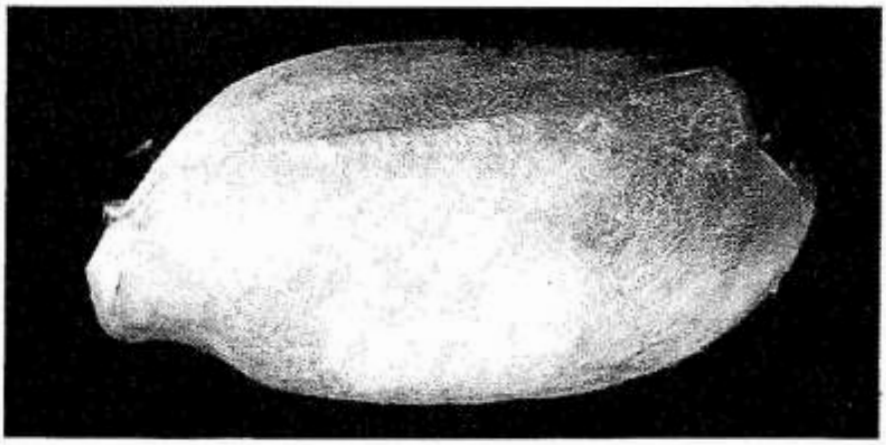

Figure 41B.
Infected groundnut seed after incubation, showing the growth of the fungus, ×7.

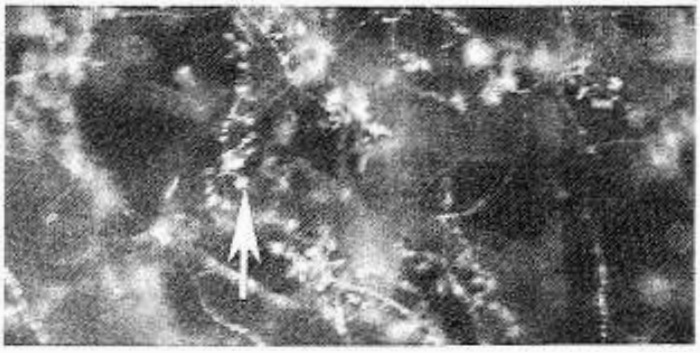

Figure 41C.
Aerial mycelium showing microconidial heads (arrowed) of F. oxysporum, *×113.*

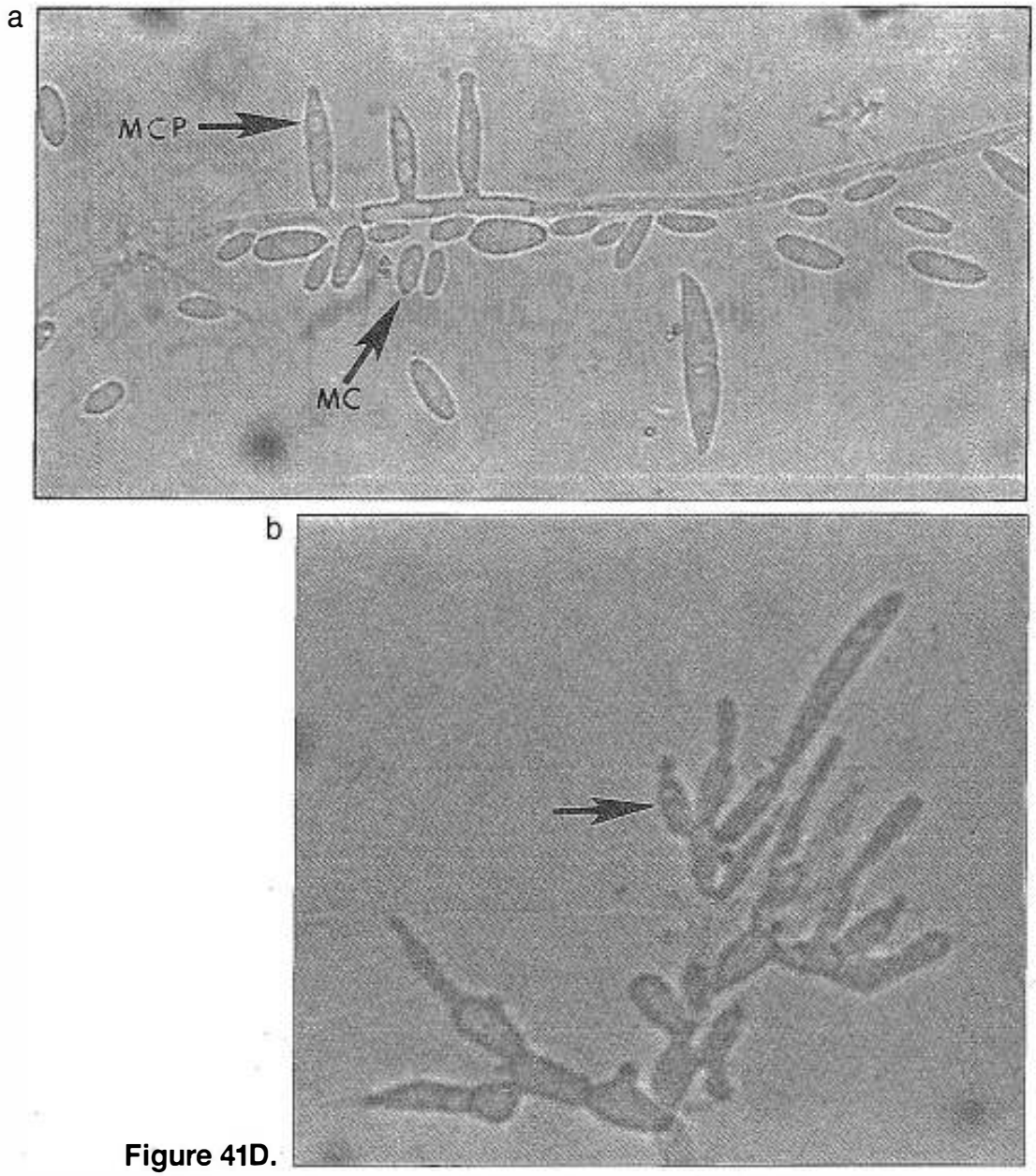

Figure 41D.
(a) Microconidiophores (MCP) and microconidia (MC), ×1130,
(b) macroconidiophores (arrowed), ×1130,

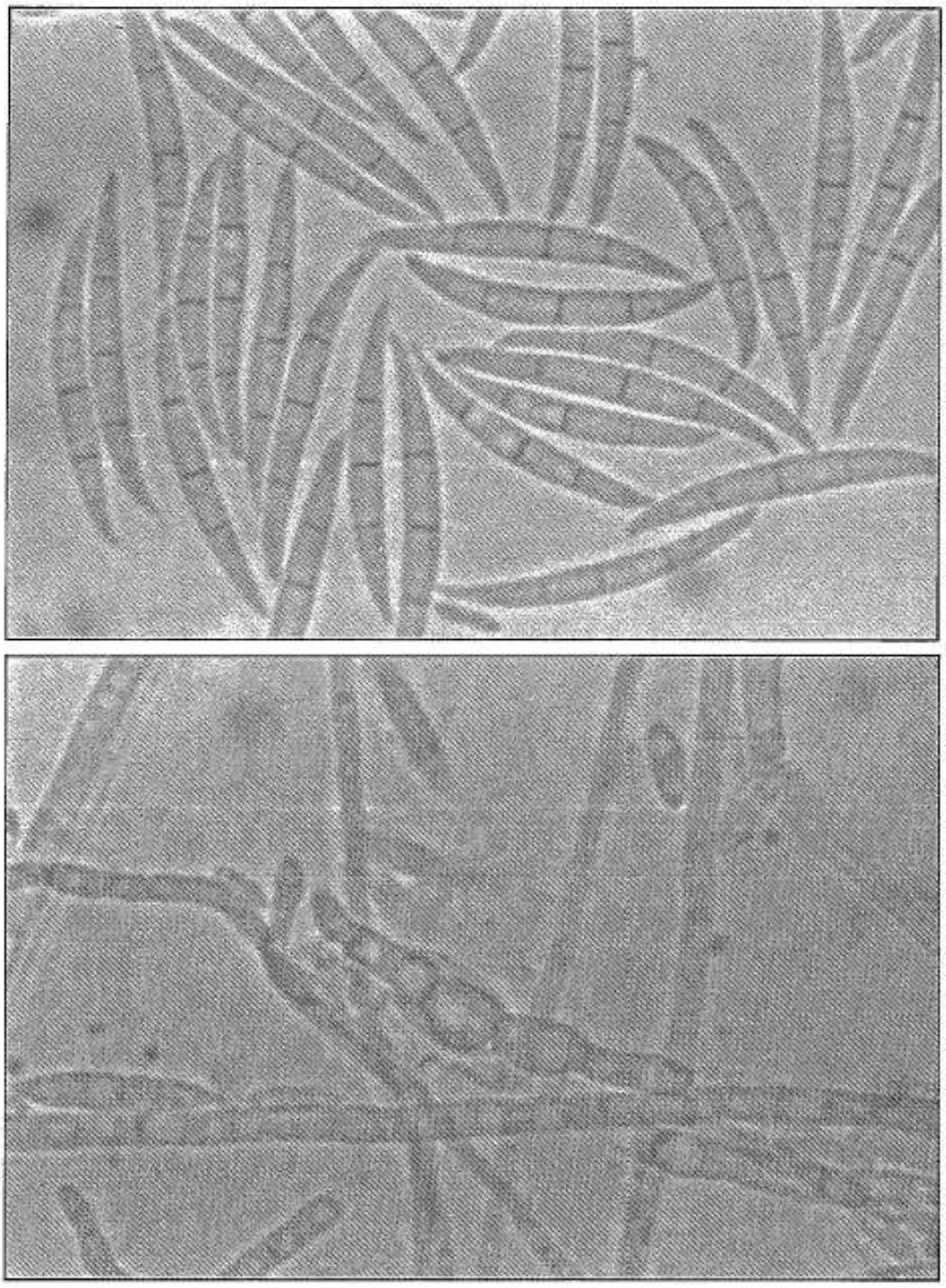

Figure 41D.
(c) macroconidia, ×1130, and (d) chlamydospores of F. oxysporum, *×1130.*

Leptosphaerulina crassiaca (Sechet.) C.R. Jackson & D.K. Bell

L. arachidicola J. Yen. Chen. & K. Huang

Leptosphaerulina crassiaca is pathogenic to groundnut and causes pepper spot/ leaf scorch diseases. The fungus usually overwinters in host debris, soil, and seed. Infected seeds can be detected by incubation tests. Numerous pseudothecia are produced on or below the seed coat of incubated seed (Fig. 42Aa). **Pseudothecia** are separate, confluent or aggregate, superficial, embedded or erumpent, often rupturing the seed coat, giving infected seed a black crusted appearance (Fig. 42Ab). They are beaked, globose, with a broad pore, and are 120–200 μm in diameter. Pseudothecia contain a few large, saccate, hyaline, thick-walled, bitunicate asci which measure 50–90 × 40–60 μm (Fig. 42Ba). Each ascus contains eight **ascospores** which are transversely and vertically septate. Ascospores are oval, clavate or ellipsoid, hyaline, often becoming slightly colored when mature, and measure 25–50 × 10–20 μm (Booth and Pirozynski 1967; Smith 1984) (Fig. 42Bb).

Control. Avoid collecting groundnut seeds from endemic fields. Information on fungicidal seed treatment to eradicate the fungus is not available.

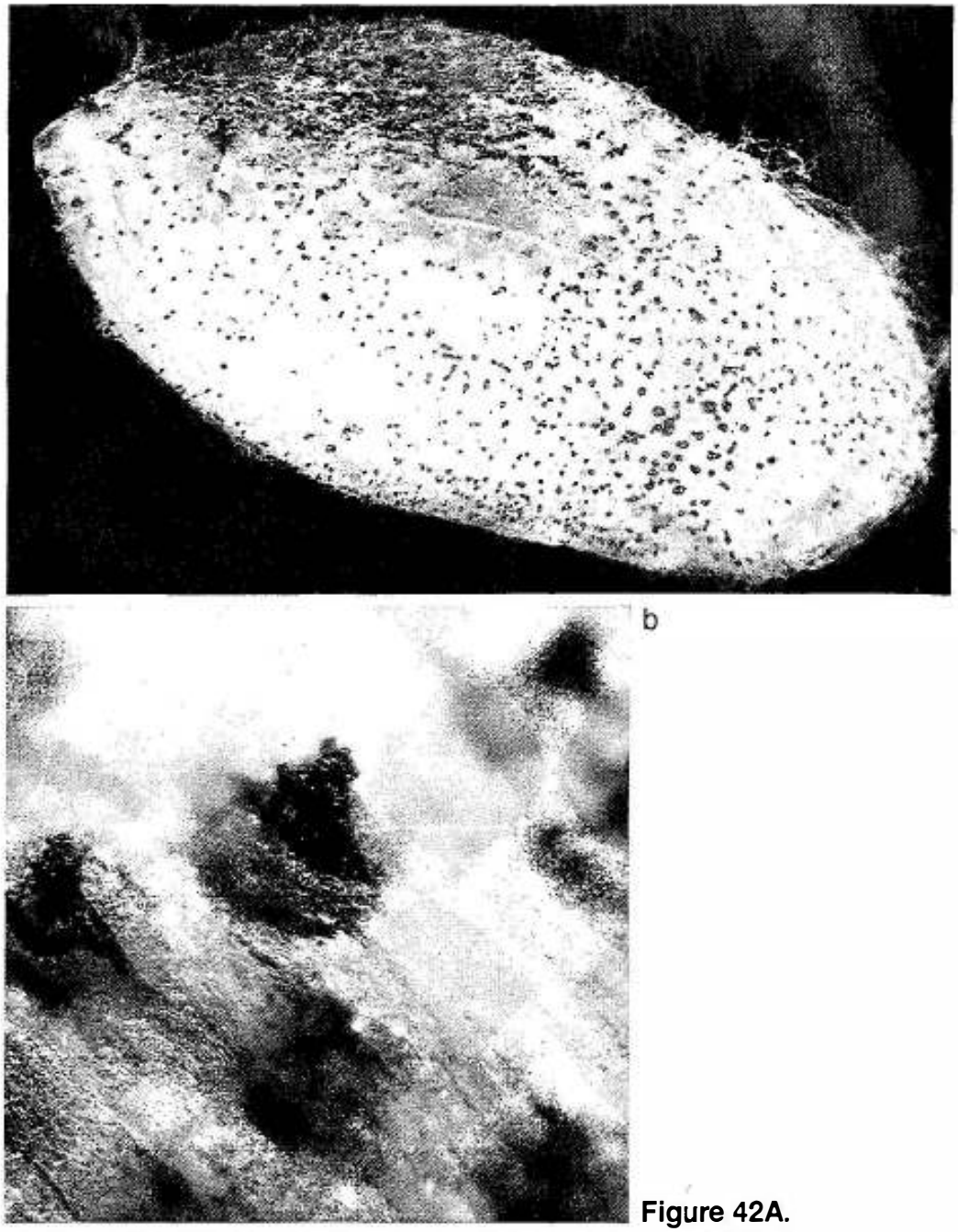

Figure 42A.
(a) Infected groundnut seed after incubation, showing the growth of Leptosphaerulina crassiaca, *×20, and (b) pseudothecia of the fungus on groundnut seed, ×113.*

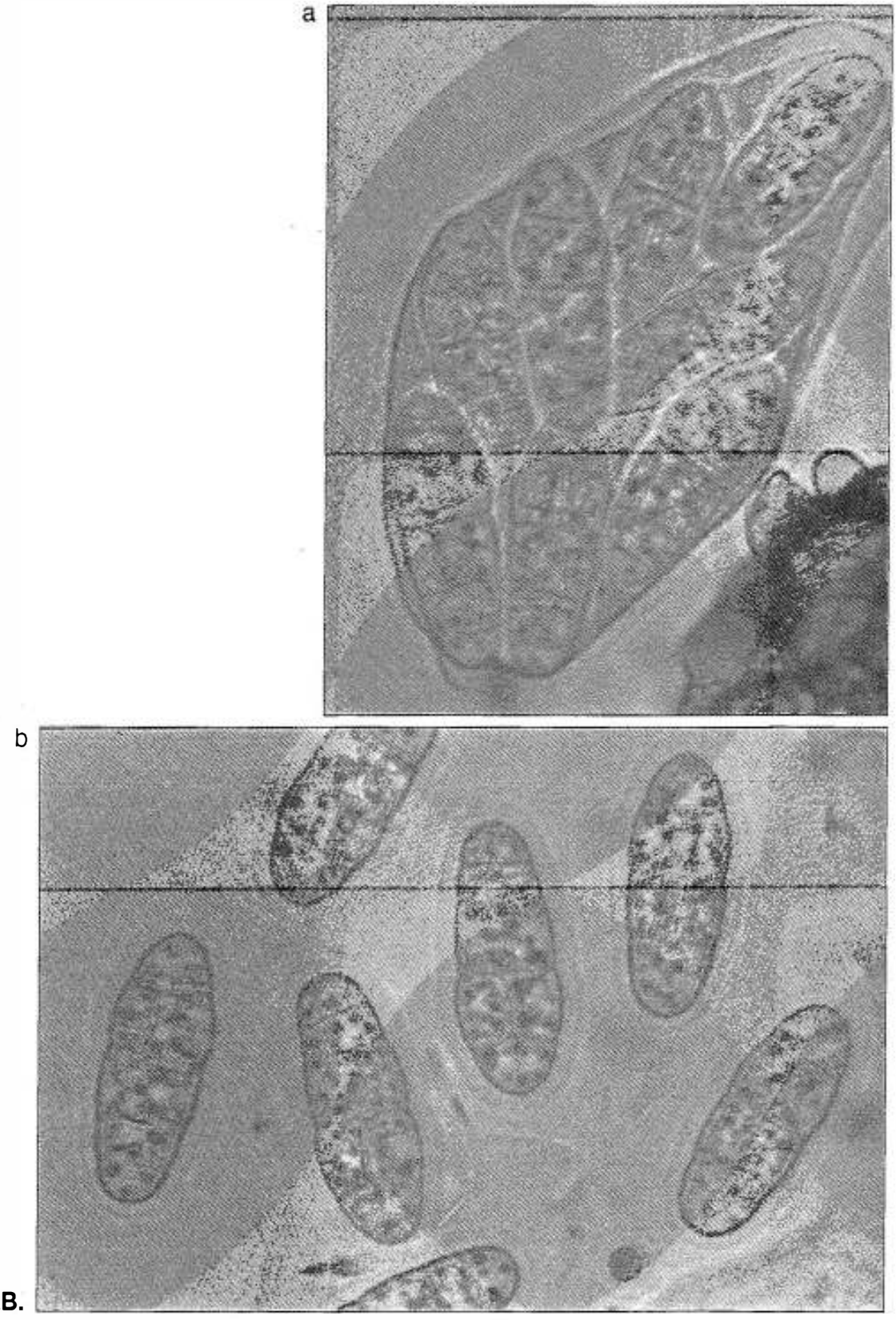

Figure 42B.
(a) Asci, ×1130, and (b) ascospores of L. crassiaca, *×1130.*

Macrophomina phaseolina (Tassi) Goidanich

Rhizoctonia bataticola (Taub.) E.J. Butler

Macrophomina phaseolina is a pathogen of chickpea, pigeonpea, and groundnut, causing root and charcoal rot diseases. The fungus survives in the seed as a dormant mycelium, sclerotia, or both. The fungus in its sclerotial state is known as *Rhizoctonia bataticola.* Seedborne infection is usually detected by incubation tests. However, in groundnut and pigeonpea, it can sometimes be detected by visual examination (Figs. 43Aa and b). The fungus may be seen as black sclerotia scattered throughout the surface of dry seed. Infected seeds are shriveled and moldy. Incubated seeds often fail to germinate; and on such seeds, the fungus produces a profuse aerial **mycelium** with pycnidia and sclerotia (Figs. 43Ba, b, and c). Hyphae are thick, gray to brown, or dark brown to black, or dull white to light brown. Sometimes the fungus grows vegetatively without producing pycnidia and sclerotia. **Pycnidia** are larger than sclerotia, dark brown to black, and scattered throughout the surface (Figs. 43Ca and inset). They may be immersed or erumpent, separate or confluent, rough, globose, or irregular, beaked, ostiolate, and 100–200 μm in diameter. When immersed, they rupture the seed coat and dehisce irregularly, giving the seed coat a charred, cancerous, or warty appearance. Sometimes the immersed pycnidia fail to emerge from the seed coat and produce olive green to dark brown, hairy hyphae around their beaks which cover the entire seed surface, giving it a cushiony velvety appearance (Fig. 43Bb). Mature pycnidia dehisce and ooze conidia in a dull white, gelatinous mass forming a cirrhus. Sometimes conidia ooze in cream-colored droplets that rest on the ostiole (Fig. 43Ca). **Conidia** are aseptate, hyaline, ellipsoid to obovoid, and 14–30 × 5–10 μm (Fig. 43D). **Sclerotia** are black, shiny, irregularly shaped, and 100 μm–1 mm in diameter (Fig. 43Cb) (Holliday and Punithalingam 1970; Kolte 1984; Nene and Reddy 1987; Reddy et al. 1990).

Control. The fungus is ubiquitous and soil reinfestation is rapid. Avoid collecting seeds for export from endemic fields. Seed dressing is useful in reducing the introduction of spores from infected seed to clean soil (Feakin 1973). For chickpea and groundnut, seed dressing with thiram at about 4 g kg^{-1} seed is advised (Suhag 1973; Lewin and Natarajan 1971). This treatment eradicates the seedborne inoculum in chickpea and in groundnut it minimizes the inoculum. To eradicate the fungus from pigeonpea seed, apply a (1:1) mixture of 30% benomyl + 30% thiram (Benlate T® at about 3 g kg^{-1} of seed) (Kannaiyan et al. 1980).

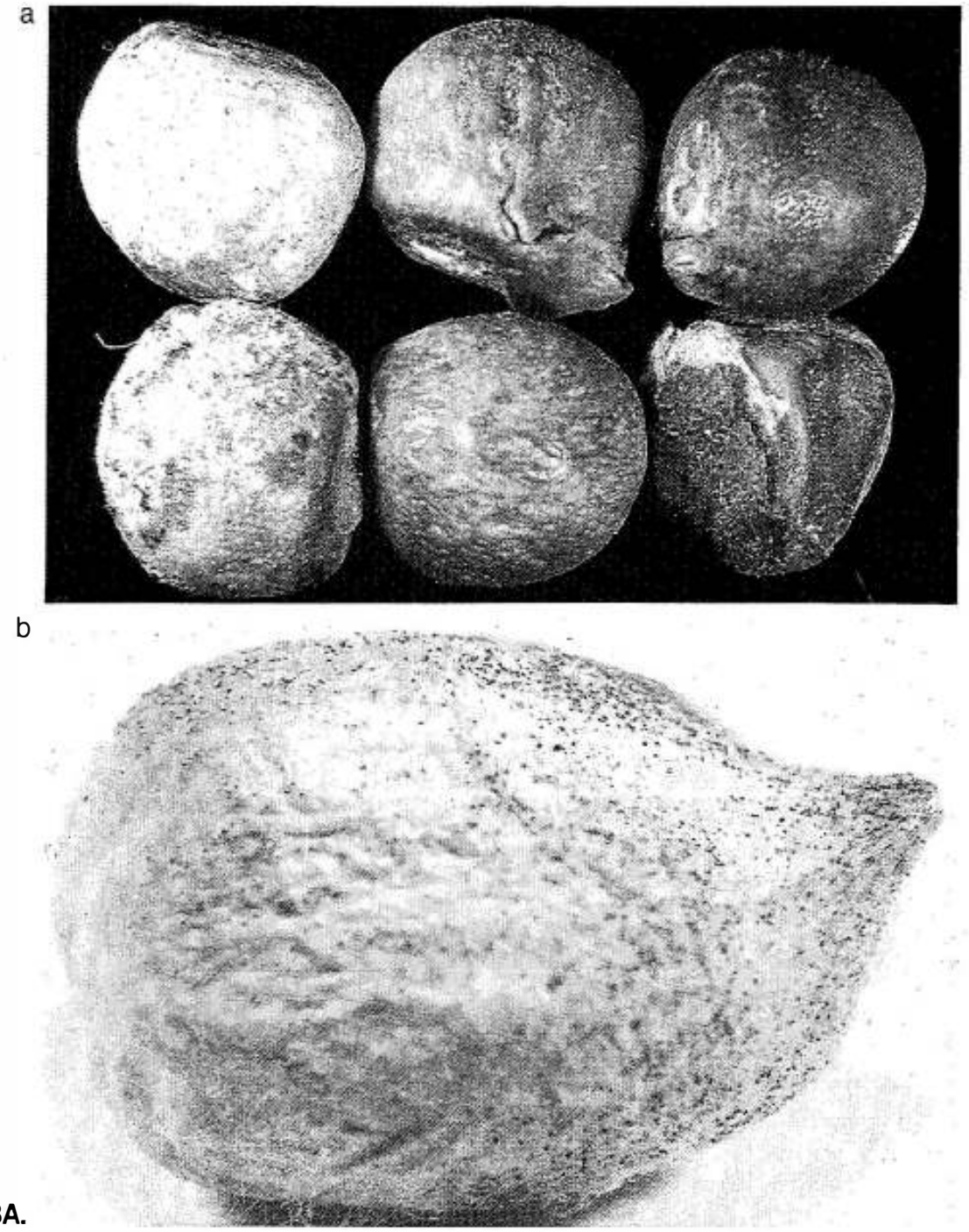

Figure 43A.
Dry seeds of: (a) pigeonpea, ×6, and (b) groundnut, ×28, showing the damage caused by Macrophomina phaseolina *(*Rhizoctonia bataticola*).*

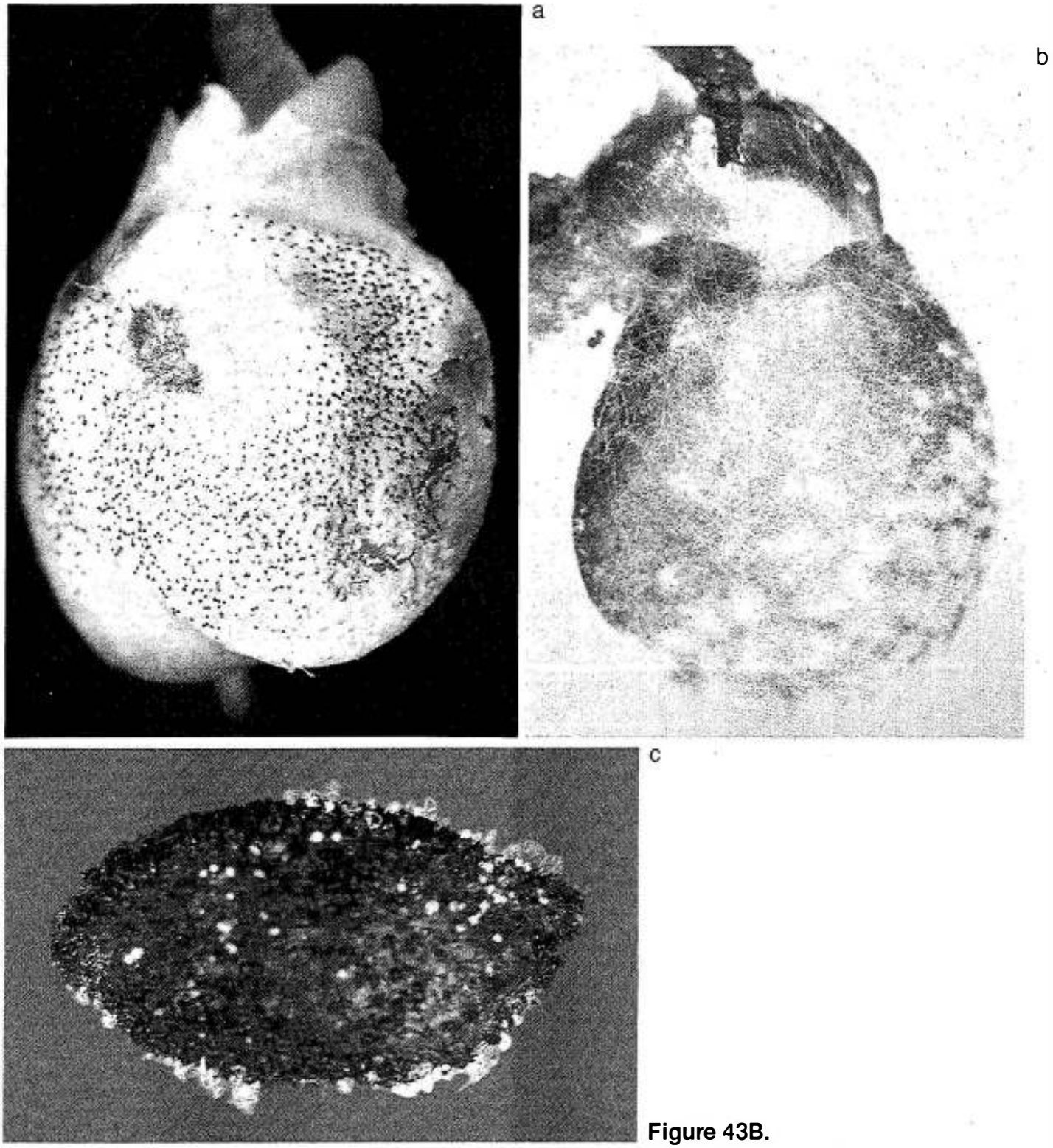

Figure 43B.
Infected seeds of: (a) chickpea, ×16, (b) pigeonpea, ×17, and (c) groundnut, ×7, after incubation, showing the growth of Macrophomina phaseolina.

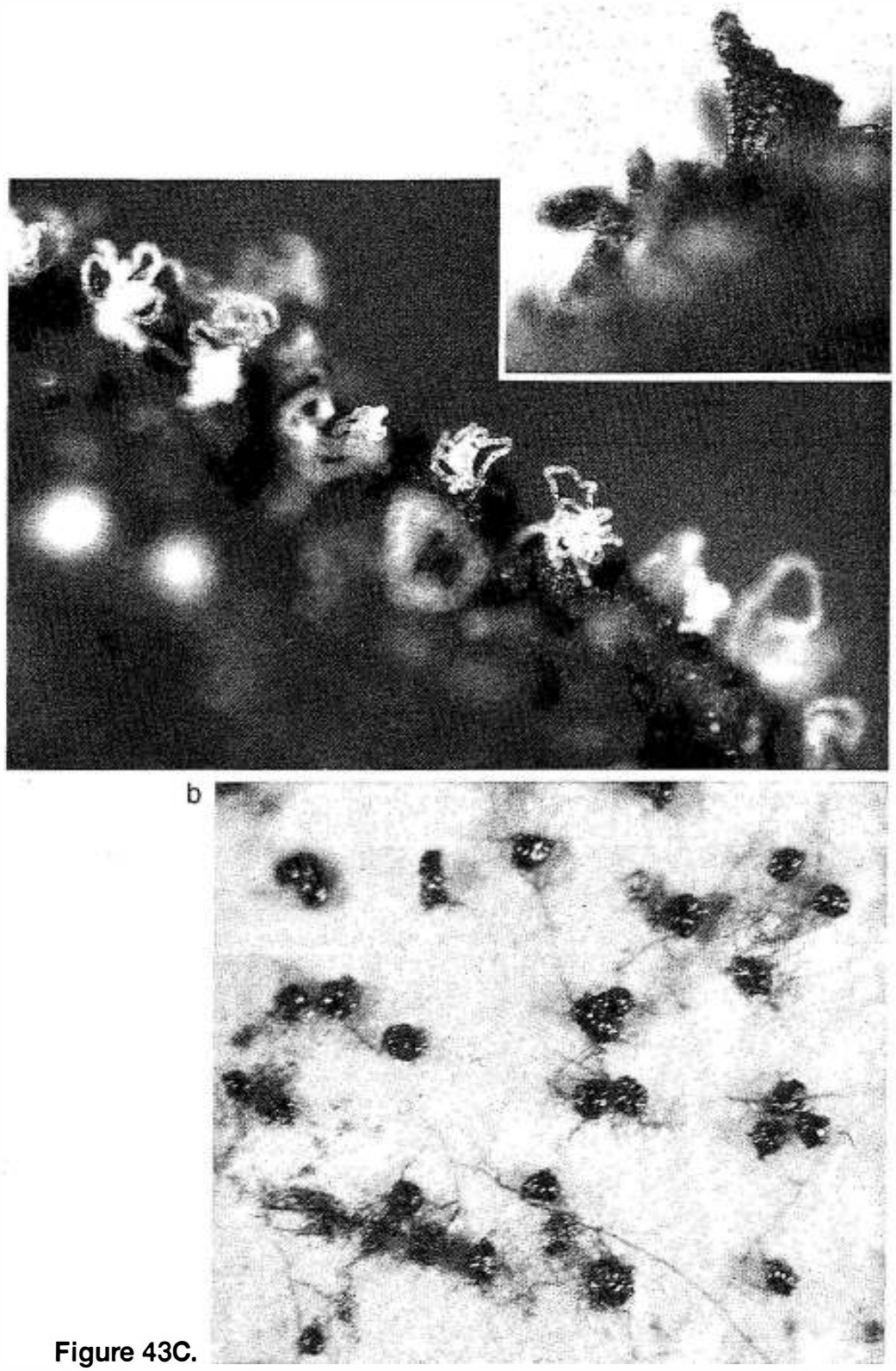

Figure 43C.
*(a) The conidial ooze, ×38; inset, pycnidia (*M. phaseolina *stage) on groundnut seed, ×113, and (b) sclerotia (*R. bataticola *stage) on chickpea seed, ×113.*

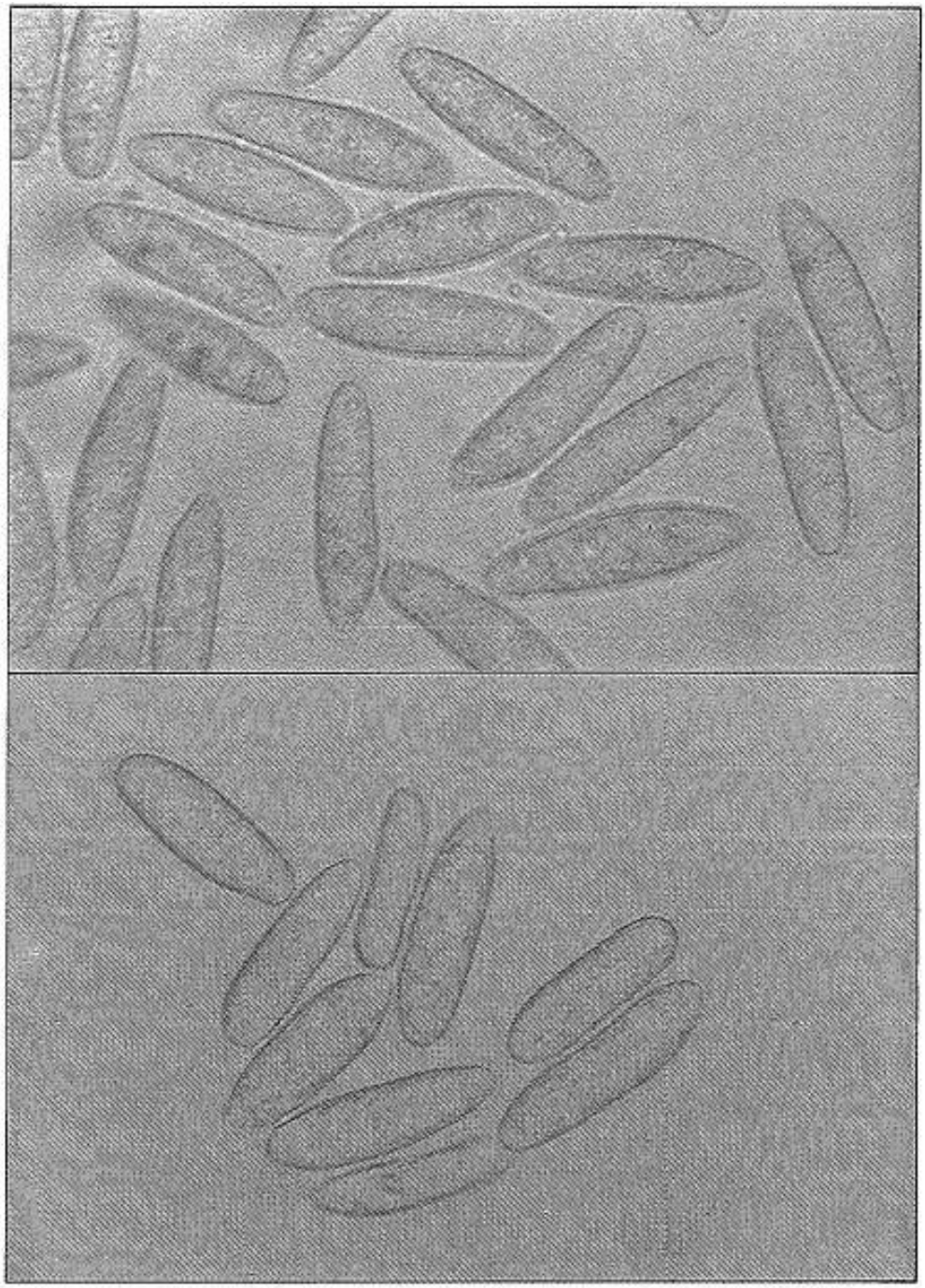

Figure 43D.
Conidia produced by M. phaseolina, *×1130.*

Rhizoctonia solani Kühn

Rhizoctonia solani attacks the root and collar regions of chickpea, pigeonpea, and groundnut, causing a variety of diseases. Since the fungus does not produce spores, it must be identified on the basis of its mycelial characteristics. It can be detected on infected seeds by incubation tests. Usually three types of growth can be seen on infected seed under a stereobinocular microscope. In the first two types, the fungus produces a thick mycelium covering the entire surface of the incubated seed and often proliferates on to the blotting paper during the blotter test (Figs. 44Aa and b). In the third type, the growth appears powdery and the mycelium is usually white to dull white, slowly turning brown as it grows (Fig. 44Ac). Hyphae are hyaline when young and become brown with age, tending to branch at right angles to parent hypha. They have a septum near the base of the branches. The hyphal portion near the septum is slightly constricted. Hyphal cells at the advancing edge of the colony are 5–12 μm wide and 250 μm long (Figs. 44Ba and b). **Sclerotia** are rarely produced on seed. They are produced slowly when colonized seed is incubated for more than 7 days up to a maximum of 21 days. Initially, sclerotia appear as shiny, guttation drops on the hyphae. They mature slowly and become brown; they are initially irregular but become globose to subglobose as they mature (Fig. 44C), consisting of pseudoparanchyma with little enclosed space (Kranz and Pucci 1963; Mordue 1974a; Nene 1980).

Control. Care should be taken not to use seed from infected crops or from areas where this fungus is known to be a problem. Information on specific seed treatment for chickpea is not available. Treating pigeonpea seeds with phenyl mercury acetate (Ceresan®) at about 3 g kg^{-1} is effective in reducing the seedborne inoculum (Kamal and Verma 1979). Benomyl at about 3 g kg^{-1} is recommended for groundnut to reduce the seedborne inoculum (Abdou and Khadr 1974).

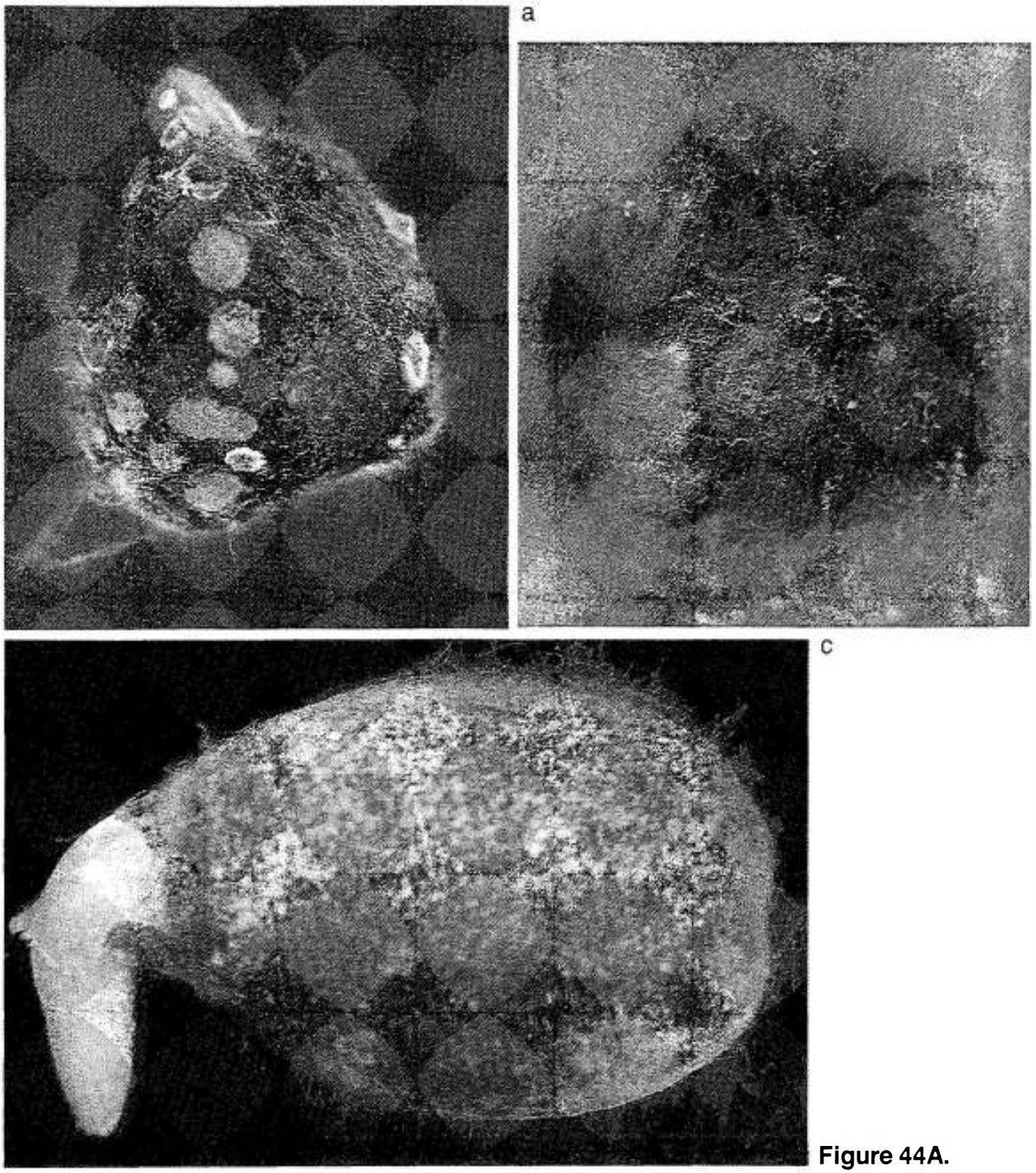

Figure 44A.
Infected chickpea seeds after incubation, showing three types of growth of Rhizoctonia solani *: (a) mycelial, ×7, (b) sclerotial, ×7, and (c) white powdery growth on groundnut, ×17.*

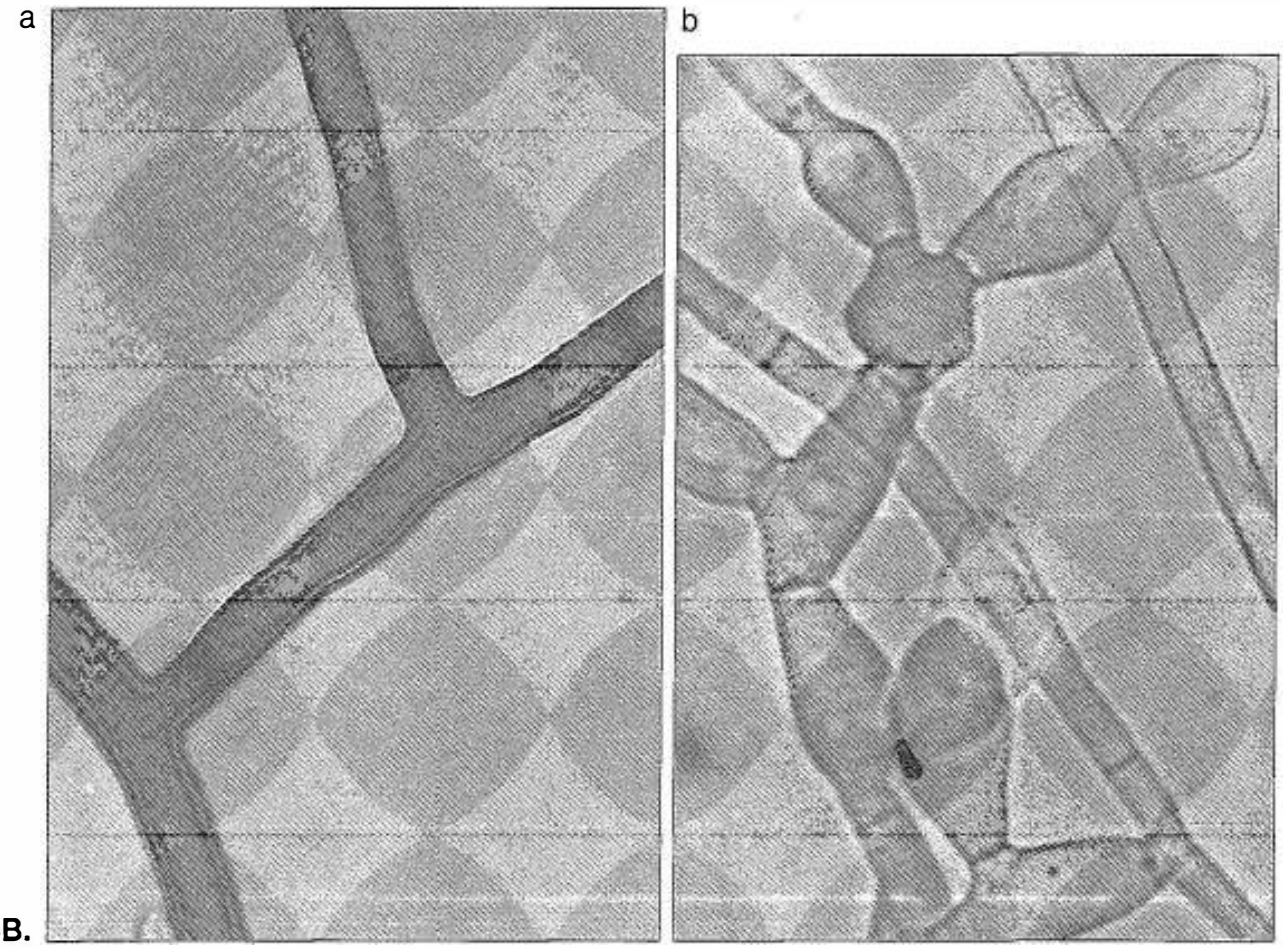

Figure 44B.
(a) Hypha, ×1130, and (b) mycelial fragments of R. solani, *×1130.*

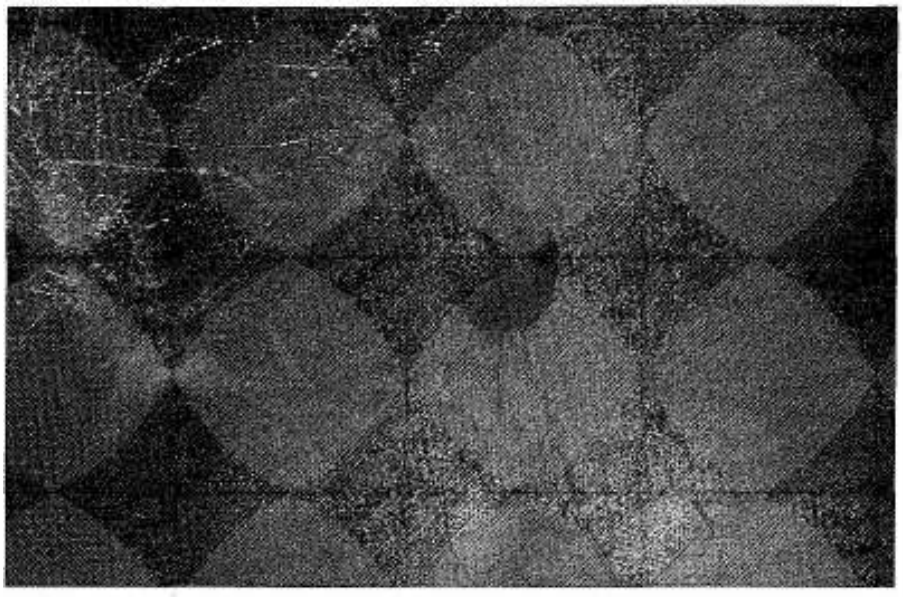

Figure 44C.
Sclerotium produced by the fungus on chickpea seed, ×75.

Sclerotium rolfsii Sacc.

Sclerotium rolfsii is an important root pathogen, causing collar rot of pigeonpea and groundnut. It also causes stem and pod rot of groundnut. The seedborne inoculum of the fungus is important in groundnut, and can be carried as admixtures of sclerotia and as dormant mycelium within the seed. Sclerotia can be detected by visual examination, and the dormant mycelium by incubation tests. On dry infected groundnut seeds, the fungus causes a discoloration called blue damage, which is characterized by the development of small spots, whose color ranges from blue to black. One such spot may be of several shades, or different spots on the same seed may be of different shades (Fig.45A). Mummified and rotted seeds also indicate the extensive damage caused by this fungus. Dark brown mustard seed like sclerotia can also be seen by visual examination. On the incubated seed, the fungus produces white radiating mycelial strands and sclerotia. The **mycelium** is very floccose, snow white, thick, cottony, and grows rapidly all over the seed and blotting paper during blotter tests. **Sclerotia,** which are the resting infective propagules of the fungus, can be seen on the mycelial strands. They are globose to ellipsoid, pinkish, buff to olive brown to clove brown. Young sclerotia appear white, producing characteristic exudate droplets around them. These sclerotia slowly become dark as they age, and resemble mustard seeds, 1–2 mm in diameter when mature (Fig.45B). Sclerotia are not formed inside the seed coat or cotyledons (Mordue 1974b; Kolte 1984; Reddy et al. 1990).

Control. Infected groundnut seed and sclerotia should be discarded during visual examination, and seed treatment with captan and quintozene mixed in 3:1 ratio or carbendazim at about 3 g kg^{-1} is advised to reduce the seedborne infection (Ghewande et al. 1987). A 1:1 mixture of 30% benomyl + 30% thiram (Benlate T® at about 3 g kg^{-1} of seed) is effective to eradicate the infection from pigeonpea seed (Kannaiyan et al. 1980).

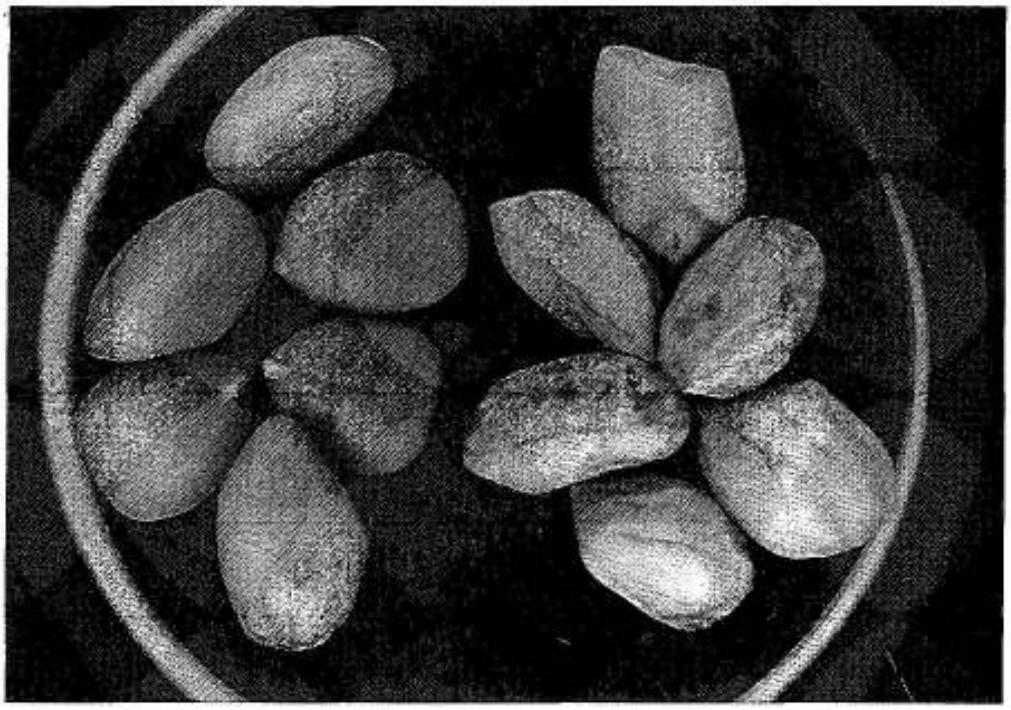

Figure 45A.
Dry seeds of groundnut showing the blue damage (right) caused by Sclerotium rolfsii*, and healthy seeds (left), ×4.*

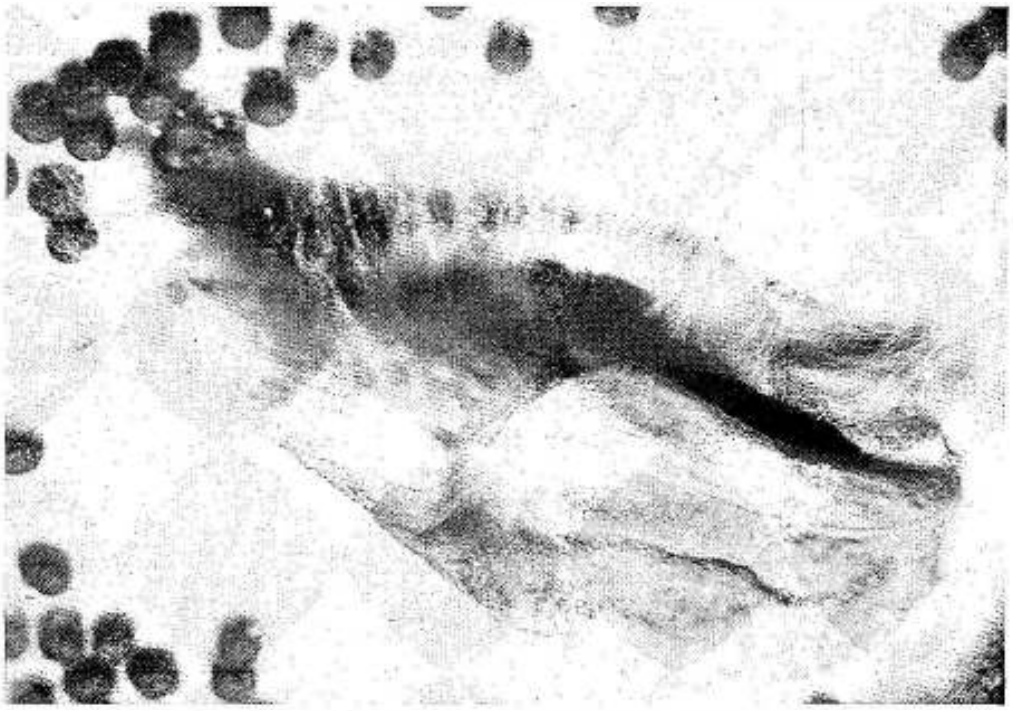

Figure 45B.
Infected groundnut seed after incubation, showing growth of the fungus, consisting of cottony white mycelial strands and mustard seed-like sclerotia, ×11.

Appendices

1. Diseases caused by seedborne fungi of ICRISAT mandate crops, and their distribution

Criteria for inclusion. (1) The report must be verifiable; that is, there must be a published account of the disease occurring in each country on the cultivated host species. Only those countries where the disease is reported on the cultivated species of ICRISAT's mandate crops have been included. (2) The economic importance or nature of infection of the pathogen on the seed (e.g., externally or internally seedborne) has not been used as a classification criterion. (3) Only seedborne fungi reported to cause disease on the host under natural conditions have been included. (4) Seedborne fungi which predominantly occur as pathogens causing grain mold have been excluded. (5) Fungi described in detail in this guide are listed in bold print in the first column. (6) In the third column the first reference to each pathogen relates to the occurrence of the disease in nature and the second reference refers to its seedborne nature: where only one reference is mentioned, it refers to both. There are three instances where these authors unpublished work is used to support the seedborne nature of the fungus, this reference is based on the official records of ICRISAT/NBPGR (National Bureau of Plant Genetic Resources) the national plant quarantine authority of India. (7) The country codes used are given below.

AF=Afghanistan
AL=Algeria
AO=Angola
AN=Antilles
AR=Argentina
AT=Australia
AU=Austria
BB=Barbados
BD=Bangladesh
BE=Belgium
BF=Burkina Faso
BG=Bulgaria
BI=Burundi
BJ=Benin
BM=Bermuda
BN=Brunei
BO=Bolivia
BR=Brazil
BT=Bhutan
BU=Myanmar
BW=Botswana
BZ=Belize
CA=Canada
CF=Central African Republic
CG=Congo
CH=Switzerland
CK=Cook Islands
CI=Canbbean Islands
CIS=Commonwealth of Independent States (earlier USSR)
CL=Chile
CM=Cameroon
CN=China
CO=Colombia
CR=Costa Rica
CS=Czechoslavakia
CU=Cuba
CY=Cyprus
DE=Germany
DK=Denmark
DO=Dominican Republic
EC=Ecuador
EG=Egypt
ES=Spain
ET=Ethiopia
FJ=Fiji
FR=France
GA=Gabon
GB=United Kingdom
GH=Ghana
GM=Gambia
GO=Grenada
GR=Greece
GT=Guatemala
GY=Guyana
HN=Honduras
HT=Haiti
HU=Hungary
IC=Ivory Coast
ID=Indonesia
IL=Israel
IN=India
IQ=Iraq
IR=Iran
IT=Italy
JM=Jamaica
JO=Jordon
JP=Japan
JW=Jawa
KE=Kenya
KM=Comoros
KR=Republic of Korea
LA=Laos
LB=Lebanon
LK=Sri Lanka
LS=Lesotho
LY=Libya
MA=Morocco
MG=Madagascar
ML=Mali
MR=Mauntania
MU=Mauntius
MW=Malawi
MX=Mexico
MY=Malaysia
MZ=Mozambique
NA=New Guinia
NB=Namibia
NC=New Caledonia
NE=Niger
NG=Nigena
NI=Nicaragua
NL=Netherlands
NP=Nepal
NU=Nive
NV=Nevis
NY=Nyasaland
NZ=New Zealand
PA=Panama
PE=Peru
PG=Papua New Giunea
PH=Philippines
PK=Pakistan
PL=Poland
PR=Puerto Rico
PY=Paraguay
PT=Portugal
RO=Romania
RW=Rwanda
SA=Saudi Arabia
SB=Solomon Islands
SD=Sudan
SL=Sierra Leone
SN=Senegal
SO=Somalia
SR=Sunname
SV=El Salvador
SY=Syna
SZ=Swaziland
TD=Chad
TG=Togo
TH=Thailand
TN=Tunisia
TO=Tonga
TR=Turkey
TT=Trinidad and Tobago
TW=Taiwan
TZ=Tanzania
UG=Uganda
US=United States of Amenca
UY=Uruguay
VE=Venezuela
VN=Vietnam
WI=West Indies
WS=Samoa
YE=Republic of Yemen
YU=Yugoslavia
ZA=South Afnca
ZM=Zambia
ZR=Zaire
ZW=Zimbabwe

Sorghum

Pathogen	Disease	Reference	Distribution
Acremonium strictum **W. gams**	Wilt	El-Shafey et al. 1979; Bandyopadhyay et al. 1987	AR, BF, CO, EG, FR, HN, IN, LS, LY, ML, MW, MX, SD, SW, TZ, US, UY, VE, ZM, ZW
Ascochyta sorghina Saccardo *A. sorghi* Sacc.	Rough leaf spot	Weimer et al. 1937; Sprague 1950	AO, BD, BM, BU, CF, CG, CN, ET, GB, GH, IT, IN, GM, JP, MW, MX, NE, NG, PA, PK, SD, SN, TD, TG, TZ, US, YE, ZM
Bipolaris maydis (Nisikado & Miyake) Shoemaker *Drechslera maydis* (Nisikado & Miyaki) Subramanian & P.C. Jain	Leaf spot	Mathur and Jai Prakash 1976; Fatima et al. 1974	BZ, IN, MW, US
Bipolaris sorghicola **(Lefebvre & Sherwin) Alcorn** *Drechslera sorghicola* (Lefebvre & Sherwin) M.J. Richardson & E.M. Fraser	Target leaf spot	Frederiksen 1986; Wu 1983	AT, BO, BW, BZ, CIS, CN, CY, EG, ET, GY, IN, IL, IC, LK, JP, MY, NG, NU, NV, PG, PH, PK, SA, SB, SD, TG, TO, TW, UG, US, VE, YE, ZW
Cercospora sorghi **Ellis & Everhart**	Gray leaf spot	Tarr 1962	AO, AR, AT, BD, BF, BI, BJ, BN, BR, BU, CF, CG, CI, CK, CM, CN, CO, CU, ES, ET, FJ, GA, GH, GM, GT, GY, HN, ID, IN, IT, JM, JP, KE, KP, KR, LA, MR, MU, MW, MX, MY, MZ, NA, NC, NE, NG, NO, NP, NU, NV, PA, PE, PG, PH, PK, PR, RW, SB, SD, SL, SN, SO, SV, SW, TD, TG, TH, TO, TT, TW, TZ, UG, US, VE, VN, WS, WI, YE, ZA, ZM, ZR, ZW
Claviceps sorghi **Kulkarni et al.**	Ergot	Kulkarni et al. 1976; Frederiksen 1986	AO, BI, BU, BW, CIS, CM, ET, GH, IN, JP, KE, LK, LS, MW, MY, MZ, NG, PH, RW, SN, SD, SO, SW, SZ, TH, TW, TZ, UG, YE, ZA, ZM, ZW
Colletotrichum falcatum Went.	Stalk rot	Van Hoof 1949	AT, BR, CN, JW, PA, US
C. graminicola **(Cesati) G.W. Wilson**	Anthracnose	Leukel et al. 1951; Basu Chaudhary and Mathur 1979	AO, AR, AT, BD, BF, BI, BO, BR, BW, BZ, CI, CN, CO, ES, ET, GT, HN, ID, IN, IT, JP, JW, KE, LS, MW, MX, MZ, NA, NE, NG, NI, NL, NR, PA, PE, PH, PK, RW, SB, SO, SD, SV, SW, TG, TH, TZ, UG, US, UY, VE, WI, YE, ZA, ZM, ZW

Sorghum continued

Sorghum *continued*

Pathogen	Disease	Reference	Distribution
Curvularia penniseti (M. Mitra) Boedijn	Leaf blight	Kore and Bhide 1975; Kavita Rani et al. 1978	IN, LK
***Exserohilum turcicum* (Pass.) K.J. Leonard & E.G. Suggs** *Helminthosporium turcicum* Pass.	Leaf blight	Mitra 1923; Wu 1983	AR, AT, BD, BF, BG, BI, BJ, BN, BO, BR, BT, BW, BZ, CA, CF, CG, CIS, CM, CN, CO, CR, CS, CU EG, ES, ET, FJ, FR, GA, GH, GM, GT, HN, ID, IL, IN, IR, IT, JP, KE, KR, LA, LS, LY, MA, ML, MW, MX, MY, NA, NE, NG, NI, NZ, PA, PE, PG, PH, PL, PR, RO, RR, RW, SA, SD, SL, SN, SO, SV, SW, SZ, TD, TG, TH, TR, TW, TZ, UG, US, VE, VN, YE, UY, ZA, ZM, ZW, ZR
Fusarium graminearum Schwabe	Stalk rot	Frederiksen 1986; Tarr 1962	AR, AT, BB, BO, CF, CL, CO, CG, EC, GA, GY, MW, PE, PY, SR, TD, UY, US, VE, ZA
***F. moniliforme* J. Sheldon**	Stalk rot	Frederiksen 1986; Mathur et al. 1975	AR, AT, BE, BF, BJ, CA, CF, CG, CU, GA, GH, HN, HU, IN, IT, KY, ML, MX, NG, PA, PH, SN, TD, TZ, US, VE, ZA, ZW
F. moniliforme J. Sheldon var. *subglutinans* Wollenw. & Reinking	Twisted top	Frederiksen 1986; Tarr 1962	AT, IN, KY, NG, PA, TZ, UG
***Gloeocercospora sorghi* Bain & Edgerton ex Deighton**	Zonate leaf spot	Bain and Edgerton 1943; Bain 1950	AO, AR, AT, BB, BF, BI, BJ, BR, BW, BZ, CF, CI, CIS, CM, CN, CO, DO, ES, ET, GA, GH, GT, HT, HN, IN, JP, KE, LS, ML, MW, MX, MY, MZ, NE, NI, NG, PA, PH, PK, RW, SD, SN, SO, SV, TD, TG, TH, TO, TW, TZ, UG, US, VE, WS, ZM, ZW
Macrophomina phaseolina (Tassi) Goidanich [*Rhizoctonia bataticola* (Taubenhaus) E.J. Butler]	Charcoal rot	Uppal et al. 1936; Kavita Rani et al. 1978	Worldwide

Sorghum continued

Sorghum *continued*

Pathogen	Disease	Reference	Distribution
Periconia circinata (L. Mangin) Sacc.	Milo disease	Leukel and Johnson 1948; Swarup et al. 1962	AT, MX, US, ZA
Peronosclerospora sorghi **(W. Weston & Uppal) C.G. Shaw.**	Downy mildew	Kulkarni 1913; Bain and Alford 1969	AO, AR, BD, BI, BO, BR, BW, BZ, CN, CO, DO, EG, ES, ET, GH, GT, HN, IL, IN, IR, JP, KE, MW, MX, MZ, NG, PA, PH, PK, PR, PY, RW, SD, SV, SW, SZ, TZ, UG, UY, US, VE, YE, ZA, ZM, ZW
Phoma sorghina **(Sacc.) Boerema, Dorenbosch & van Kesteren** *Phoma insidiosa* Tassi	Leaf spot	Tarr 1962; Bhale and Khare 1982	AR, BE, BO, BR, BU, BW, CA, CF, CG, CI, CIS, CN, ET, GA, IN, IT, KE, MW, MY, NE, NG, NY, PG, PR, PT, SD, SN, TD, TZ, TW, UG, US, ZA, ZM, ZW
Ramulispora sorghi (Ellis & Everhart) Olive & Lefebvre	Sooty stripe	Olive et al. 1946; Frederiksen 1986	AR, AT, BF, BI, BR, BW, CF, CG, CN, ET, GA, HN, HT, IN, JP, KE, ML, MW, MX, MZ, NE, NG, RW, SD, SD, SN, SO, SV, SW, TD, TW, TZ, US, YE, ZM, ZW
Sporisorium cruentum **(Kühn) K. Vánky** *Sphacelotheca cruenta* (Kuhn) A.A. Potter	Loose smut	Tarr 1962	AF, AR, BB, BE, BI, BG, BR, BU, BW, CA, CF, CG, CI, CIS, CM, CN, CS, CU, CY, DE, ES, ET, GA, GH, GR, HN, HT, HU, IL, IN, IQ, IR, IT, JM, JP, KE, KR, LB, MA, MU, MW, MX, MY, MZ, NE, NG, NI, PK, PL, PR, RO, RW, SD, SN, SO, SV, SW, TD, TN, TR, TW, TZ, UG, US, VE, YE, YU, ZA, ZM, ZR, ZW
Sporisorium holci-sorghi **(Rivolta) K. Vánky** *Sporisorium reilianum* (Kühn) McAlpine *Sphacelotheca reiliana* (Kuhn) G.P. Clinton	Head smut	Tarr 1962	AR, AT, BF, BG, BI, BR, BT, BU, BW, CG, CI, CIS, CL, CM, CN, CS, CY, EG, ES, ET, FR, GA, GH, GT, DE, HN, ID, IL, IN, IR, IT, JP, JM, JW, KE, KR, LS, MA, ML, MR, MW, MX, MY, MZ, NE, NG, PA, PG, PK, RO, RW, SD, SN, SO, SV, SW, SZ, TD, TG, TW, TZ, UG, US, YE, ZA, ZM, ZW, ZR

Sorghum continued

Sorghum *continued*

Pathogen	Disease	Reference	Distribution
Sporisorium sorghi **Link** *Sphacelotheca sorghi* (Link) G.P. Clinton	Kernel smut	Tarr 1962	AF, AN, AO, AR, AT, AU, BD, BE, BF, BG, BI, BM, BN, BO, BR, BU, BW, CA, CF, CG, CH, CI, CIS, CL, CM, CN, CO, CR, CS, CU, CY, DE, DK, EG, ES, ET, FR, GA, GB, GH, GR, GT, HN, HT, HU, IL, IN, IQ, IR, IT, JM, JP, KE, KR, LB, LK, LS, MA, ML, MW, MX, MY, MZ, NE, NG, NI, NL, NV, NY, NZ, PA, PE, PG, PH, PK, PR, PT, RW, RO, SA, SB, SD, SL, SN, SO, SV, SW, TG, TR, TW, TZ, UG, US, VE, VN, WI, YE, YU, ZA, ZM, ZR, ZW
Tolyposporium ehrenbergii **(Kühn) Patouillard**	Long smut	Tarr 1962; Manzo 1976	BF, BI, BW, CIS, CM, CN, EG, ET, GH, GM, IL, IN, IQ, IR, JP, KE, ML, MR, MW, NE, NG, PK, RW, SA, SD, SN, SO, TR, TZ, UG, YE, ZA, ZM , ZW

Continued

Pearl Millet

Pathogen	Disease	Reference	Distribution
Bipolaris sacchari **(E.J. Butler) Shoem.** *Drechslera sacchari* (E.J. Butler) Subramanian & P.C. Jain	Leaf spot	Misra et al. 1974; Ravinder Reddy et al. 1990	IN, US
Bipolaris setariae **(Sawada) Shoemaker** *Drechslera setariae* (Saw.) Subramanian & P.C. Jain	Leaf spot	Wells 1967; Wells and Winstead 1965	IN, US
Claviceps fusiformis **Loveless**	Ergot	Ramakrishnan 1963; Thakur et al. 1984	AO, BF, BW, ET, GH, GM, IN, MW, NE, NG, SD, SO, SN, TZ, UG, YE, ZM, ZW
Curvularia penniseti **(M. Mitra) Boedijn**	Leaf spot	Misra 1959	IN
Fusarium moniliforme J. Sheldon	Top rot/ twisted top	Ramakrishnan 1941; Konde et al. 1980	IN, NG, US
Pyricularia penniseti **Prasada & Goyal**	Brown leaf spot	Prasada and Goyal 1970; Singh and Pavgi 1977	CN, PK, IN
Sclerospora graminicola **(Sacc.) J. Schroet**	Downy mildew	Kulkarni 1913; Shetty et al. 1980	AR, BF, BW, CM, GA, GH, GM, IL, IN, ML, MW, MZ, NE, NG, NL, PK, SA, SD, SN, TD, TZ, YE, ZA, ZM, ZW
Tolyposporium penicillariae **Bref.**	Smut	Ajrekar and Likhite 1933; Thakur and King 1988	AO, AT, BF, BU, BW, CM, EG, GH, GM, IC, IN, IQ, KE, ML, MW, MZ, NB, NE, NG, NY, PK, SN, SL, SD, TG, TD, TZ, UG, US, YE, ZM, ZR, ZW

Continued

Finger Millet

Pathogen	Disease	Reference	Distribution
Bipolaris nodulosa **(Berk. & M.A. Curtis) Shoemaker**	Leaf spot/ blight	Mitra and Mehta 1934; Grewal and Mahendrapal 1965	IN, MU, MW, NP, PH, TZ, UG, US, ZM, ZW
Bipolaris setariae **(Sawada) Shoemaker** *Drechslera setariae* (Sawada) Subramanian & P.C. Jain	Leaf blight	Hiremath and Sulladmath 1985; K.M. Ahmed and Ch. Ravinder Reddy, unpublished.	IN
Melanopsichium eleusinis **(Kulkarni) Mundkur & Thirumalachar** *Ustilago eleusinis* Kulkarni	Smut	Kulkarni 1922; Ramakrishnan 1963	AT, IN, TZ
Pyricularia setariae **(Wallace) Ramakrishnan** *Pyricularia grisea* (Cooke) Sacc.	Leaf and neck blast	Ramakrishnan 1963; Shetty et al. 1985	CF, IN, JP, KE, MW, MY, NP, SO, TZ, UG, US, ZM
Sclerophthora macrospora **(Sacc.) Thirum., Shaw & Naras**	Downy mildew	Venkataraÿan 1946; Ramakrishnan 1963	IN, US

Continued

Chickpea

Pathogen	Disease	Reference	Distribution
Alternaria alternata **(Fr.) Keissler**	Leaf blight	Vishwakarma and Chaudhry 1974; Haware et al. 1986	BD, HU, IN
Ascochyta pinodes **L.K. Jones** [*Mycosphaerella pinodes* (Berk. & Bloxam) Vestergr.]	Blight	Bretag and Mebalds 1987	AT, HU, IN
Ascochyta rabiei **(Pass.) Labrousse** *Phoma rabiei* (Pass.) Khune & J.N. Kapoor	Blight	Labrousse 1931; Maden et al. 1975	AL, AT, BD, BG, CA, CIS, CO, CY, EG, ES, ET, FR, GR, HU, IL, IN, IQ, IR, IT, JO, LB, MA, MX, PT, PK, RO, SD, SY, TR, TN, TZ, US
Botrytis cinerea **Pers. ex Fr.**	Gray mold	Joshi and Singh 1969; Laha and Grewal 1983	AR, AT, BD, CA, CO, IN, NP, PK, ES, TR, US
Colletotrichum dematium **(Pers. ex Fr.) Grove**	Blight	Mishra et al. 1975; Haware et al. 1986	IN
Fusarium oxysporum **Schlechtend emend. Snyd. Hans f. sp. *ciceri*** **(Padwick) Snyd. & Hans**	Wilt	Conci et al 1985; Haware et al. 1986	AL, AR, AT, BD, BU, CL, CO, ET, ES, HU, IL, IN, IR, IQ, IT, KE, LB, MA, MW, MX, NP, PE, PK, SD, SY, TN, US
F. solani **(Mart.) Sacc.**	Root rot/ wilt	Grewal et al. 1974; Maden 1987	AR, CL, ES, HU, IN, MX, SY, US
Neocosmospora vasinfecta **E.F.Smith**	Wilt	Haware and Nene 1976; K.M. Ahmed and Ch. Ravinder Reddy, unpublished	IN
Phoma medicaginis **Malbr. & Roum.**	Blight	Haware and Nene 1981	AT, BD, HU, IN, US
Rhizoctonia bataticola **(Taubenhaus) E.J. Butler** [*Macrophomina phaseolina* (Tassi) Goidanich]	Dry root rot	Dastur 1935; Suhag 1973	AT, BD, ES, ET, IN, IR, KE, LB, MX, PK, SY, US
Rhizoctonia solani **Kühn**	Wet root rot	Nene and Reddy 1987; Maden 1987	AR, AT, BD, CL, ET, IN, IR, MA, MX, PK, SY, TR, US
Stemphylium sarciniforme (Cavara) Wiltshire	Leaf spot	Das and Sen Gupta 1961; Kaiser 1972	HU, IN, IR, SY

Continued

Pigeonpea

Pathogen	Disease	Reference	Distribution
***Alternaria alternata* (Fr.) Keissler**	Leaf spot/ blight	Venkateswarulu et al. 1981; Lokesh et al. 1987	IN
A. tenuissima (Kunze ex Pers.) Witshire	Leaf spot	Kannaiyan and Nene 1977; Kamal and Verma 1979	IN, PR, US, ZM
***Botryodiplodia* sp**	Blight	Nene et al. 1989; Hepperly and Rudriguez 1984	PR, IN
Botrytis cinerea Pers. ex Fr.	Blight	Nene et al. 1989; Kamal and Verma 1979	BD, NP
***Fusarium udum* Butler**	Wilt	Butler 1910; Kannaiyan et al. 1980	BD, GH, GO, ID, IN, KE, MW, MU, NP, NV, TH, TT, TZ, UG, VE, ZM
F. semitectum Berk. & Ravenel	Blight	Singh 1988; Ellis and Paschal 1979	IN
Phoma sp	Blight/Leaf spot	Lo'pez Rosa 1969	DO, IN, MW, PA, PR, TT, ZM
Phomopsis sp	Leaf spot	Nene et al. 1989; Hepperly and Rodriguez 1984	PR
***Phyllosticta cajani* Rangel**	Leaf spot	Khune and Kapoor 1981; Kumud Kumar 1980	BR, IN, JM, PR, TT, US, WI, SL, SM
***Rhizoctonia bataticola* (Taubenhaus) E.J. Butler** [*Macrophomina phaseolina* (Tassi) Goidanich]	Root rot	Rathi and Lal 1977; Kannaiyan et al. 1980	IN, JM, NP, TT, US, MW, ZM
***R. solani* Kühn**	Root rot	Dwivedi and Saksena 1975; Kamal and Verma 1979	IN, ML, PH, SL, TT, UG, ZM
Sclerotium rolfsii Sacc.	Collar rot	Phelps et al. 1974; Kannaiyan et al. 1980	IN, PK, PR, TT, US, VE, ZM

Continued

Groundnut

Pathogen	Disease	Reference	Distribution
Alternaria alternata **(Fr.) Keissler**	Leaf blight	Subrahmanyam et al. 1981; Joffe and Borut 1966	BB, IN, NI, SD, TH, UG, US, VN, ZM
Aspergillus flavus **Link ex Fr.**	Aflaroot/ yellow mold	Chohan and Gupta 1968; Kolte 1984	Worldwide
A. niger **v.Tieghem**	Crown rot/ collar rot	Jackson 1962; Jackson and Bell 1969	Worldwide
Botryodiplodia theobromae **Pat.** *Lasiodiplodia theobromae* (Pat.) Griffon & Maubl.	Collar rot	Orian 1949; Mercer and Kisyombe 1978	AT, BR, JP, IL, IN, TH, US, VE, ZA, ZM
Botrytis cinerea Pers ex Fr.	Blight/gray mold	Wu 1979; Figueiredo and Cardoso Rosa 1968	AT, BR, CIS, JP, MW, NY, RO, TW, TZ, US, VE, VN, ZA, ZM, ZW
Colletotrichum dematium **(Pers. ex Fr.) Grove**	Anthracnose	Saksena et al. 1967; Ravinder Reddy et al. 1990	AR, IN, NE, NG, PA, TH, TW, TZ, UG, US
Cylindrocladium crotalariae (C.A Loos) D K Bell & Sobers	Cylindrocladium black rot	Porter and Mozingo 1986	AT, CN, IN, JP, US
Fusarium equiseti **(Corda) Sacc.**	Leaf spot	Ghewande Raj Jhala et al. 1987; El-Magraby and El-Maraghy 1988	IN, ZM
F. oxysporum **Schlechtend; emend Sny. & Hans.**	Damping off/ wilt/root rot	Ghewande et al. 1985; ElMagraby and El-Maraghy 1988	Worldwide
F. solani **(Mart.) Sacc.**	Wilt	Lima and Aguillar 1978; Mercer and Kisyombe 1978	BR, IN, MW, US, ZM
Leptosphaerulina crassiaca **(Sechet) C.R. Jackson & D.K. Bell**	Leaf scorch pepper leaf spot	Nayudu 1963; K.M. Ahmed and Ch. Ravinder Reddy, unpublished	AO, AR, AU, BF, CA, IN, MG, MU, MW, MX, MZ, NE, NG, SN, SZ, TW, TH, US, VN, ZM, ZW
Neocosmospora vasinfecta E F. Smith	Wilt	Beard and Van Wyk 1985; Barbosa Maria 1965	US, ZA, ZM

Groundnut continued

Groundnut

Pathogen	Disease	Reference	Distribution
Puccinia arachidis[1] Spegazzini	Rust	Subrahmanyam and McDonald 1982; Peregrine 1971	Worldwide
Rhizoctonia bataticola **(Taubenhaus) E.J. Buttler** [*Macrophomina phaseolina* (Tassi) Goidanich]	Root rot/ stem rot/ wilt/ stem blight/ dry rot	Garren and Wilson 1951; Mridha and Fakir 1978	Worldwide
R. solani **Kühn**	Root rot/ pod rot	Jackson and Bell 1969; Garren and Higgins 1947	Worldwide
Sclerotina minor Jagger	Sclerotinia blight	Porter and Beute 1974; Wadsworth and Melouk 1985	AR, AT, BR, IL, ZA, US
Sclerotium rolfsii **Sacc.**	Stem rot/ southern blight/white mold	Alstatt 1944; Mridha and Fakir 1978	Worldwide
Chalara elegans Nag Raj & Kendrick *Thielaviopsis basicola* (Berk & Broome) Ferraris	Black hull	Labuschagne et al. 1980; Frank 1986	AR, IL, IT, ZA, US

1. Quarantine important when vegetatively propagating material is used for export.

2. International Seed Health Testing Methods (ISHTM) used by ICRISAT to detect seedborne fungi

There are many types of ISHTM. The ones most commonly used at ICRISAT are described below.

Dry seed examination. Dry seed are inspected for the presence of admixtures such as ergot or other sclerotia, free insects, nematode galls, smutted kernels, discolorations, and blemishes due to pathogenic organisms. An illuminated swinging-arm desk magnifier of ×2 magnification is used to examine the seed. Sometimes a stereoscopic microscope is also useful to detect fungal fructifications or resting hyphae on the seed. Impurities and admixtures observed in seed lots are readily distinguished as inert matter by seed analysts. A seed pathologist should be able to identify specific problems.

Seed-washing test. Spores of some seedborne fungi adhere to the seed surface and are not revealed during either visual or incubation tests, thus escaping detection. To detect such seedborne fungi, a sedimentation or seed-washing test has proved useful in detecting the spores of fungi causing downy mildew, rust, and smut diseases. About 50 seeds are drawn randomly into a test tube containing 10 mL of distilled water and a few drops (10–20) of 95% ethyl alcohol or a detergent. The test tube containing the seed is placed on a mechanical shaker for 10 min. The suspension is then centrifuged at 3000 rpm for 10 min. After discarding the supernatant, the pellet is resuspended in 2 mL of sterile water. A few drops of this suspension are examined for the presence of fungal spores under a compound microscope. Several mounts of the suspension should be made so that pathologists can use them to identify spores they contain.

Blotter test. Several methods of incubation can be used to observe seed mycoflora. This standard blotter method of seed health testing is most widely used for phytosanitary certifications because it is easy to conduct, economical, and suitable for the detection of a large variety of seedborne fungi. In this method, seeds are sown in 100 × 25 mm petri dishes containing three layers of moistened absorbent (blotting) paper. Ten seeds each of sorghum, pearl millet, and finger millet are sown and five seeds each of chickpea, pigeonpea, or groundnut. The seeds are arranged in the petri dishes equidistant from one another and incubated at 22 ± 2°C, under near ultraviolet (NUV) light, with 12-h alternate cycles of light and dark for 7 days. After incubation, the seeds in each petri dish are examined under a stereobinocular microscope. The characteristics of the fungal colonies observed can be used to identify specific pathogens.

3. Fungicidal seed dressing schedule used at ICRISAT Center

Crop	Chemical	Dosage (g kg^{-1} seed)	Reference
Sorghum	benomyl + thiram (1:1)	2.0	Vidyasekaran 1983
	metalaxyl	1	Anahosur and Patil 1980
Pearl millet	benomyl + thiram (1:1)	2.5	Ravinder Reddy et al. 1990
	metalaxyl[1]	2	India 1977
Finger millet	thiram + carbendazim (1:1)	2.5	Dutta and Jha 1983
Chickpea	benomyl + thiram (1:1)	0.90	Haware et al. 1978
	thiabendazole	1.5	Reddy and Kababeh 1984
Pigeonpea	benomyl + thiram (1:1)	0.90	Kannaiyan et al. 1980
Groundnut	benomyl and thiram	2.5	Shekhawat et al. 1986
		3.3	Lewin and Natarajan 1971

1. • Soak the seeds in 0.1% mercuric chloride for 10 min and then wash thoroughly in running water for 5 min.
   • Transfer the seeds immediately into a water bath set at 55°C for 12 min.
   • Transfer the seeds to water at room temperature for a few minutes, and then transfer them to an incubator set at 35°C for 12 h and then at 40°C for 1 h.
   • Seeds should then be immersed in a suspension containing 2 g of metalaxyl in 800 mL of 1% acqueous methyl cellulose for 5–6 h. This is sufficient for 1 kg of dry pearl millet seeds.
   • The treated seeds must be dried under shade/sunlight. Seeds treated this way can be sown immediately or up to 4 months after the treatment.

References

References

Abdou, Y.A., and **Khadr, A.S.** 1974. Systemic control of seedling and pod rot disease of peanuts (*Arachis hypogaea*). Plant Disease Reporter 58(2):176–179.

Agarwal, S.C., and **Khare, M.N.** 1978. Chemical control of seed-borne pathogenic fungi of jowar. Pesticides 12(9):25–26.

Ainsworth, G.C. 1965a. *Sphacelotheca cruenta*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 71.

Ainsworth, G.C. 1965b. *Sphacelotheca reiliana*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 73.

Ainsworth, G.C. 1965c. *Sphacelotheca sorghi*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 74.

Ainsworth, G.C. 1965d. *Tolyposporium ehrenbergii*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 76.

Ainsworth, G.C. 1965e. *Tolyposporium penicillariae*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 77.

Ainsworth, G.C., James, P.W., and **Hawksworth, D.L.** 1971. Ainsworth and Bisby's dictionary of the fungi. Sixth edn. Kew, Surrey, UK: Commonwealth Mycological Institute. 663 pp.

Ajrekar, S.L., and **Likhite, V.N.** 1933. Observations on *Tolyposporium penicillariae* Bref. (the bajri smut fungus). Current Science 1(7):215.

Alstatt, G.E. 1944. *Sclerotium rolfsii* on peanuts in Texas. Plant Disease Reporter 28(9):1097.

Anahosur, K.H., and **Hegde, R.K.** 1980. Head molds of sorghum, their effect on seed germination and in vitro evaluation of fungicides. Mysore Journal of Agricultural Sciences 14(1):60–63.

Anahosur,K.H., and **Patil, S.H.** 1980. Chemical control of sorghum downy mildew in India. Plant Disease 64(11):1004–1006.

Atac, A. 1989. Effectiveness of some fungicides against covered kernel smut disease [*Sphacelotheca sorghi* (Link) Clint.] on sorghum [*Sorghum vulgare* Pers. var. *technicum* (Koern) Jav] in Cukurova region. Journal of Turkish Phytopathology 18:(1–2)47–50.

Bain, D.C. 1950. Fungi recovered from seed of *Sorghum vulgare* Pers. Phytopathology 40(5):521–522.

Bain, D.C., and **Alford, W. W.** 1969. Evidence that downy mildew (*Sclerospora sorghi*) of sorghum is seedborne. Plant Disease Reporter 53(10):802–803.

Bain, D.C., and **Edgerton, C.W.** 1943. The zonate leaf spot, a new disease of sorghum. Phytopathology 33(3):220–226.

Bandyopadhyay, R., Mughogho, L.K., and **Satyanarayana, M.V.** 1987. Systemic infection of sorghum by *Acremonium strictum* and its transmission through seed. Plant Disease 71(7):647–650.

Barbosa Maria, A.De F. 1965. A new species of *Neocosmospora* found in stored peanut. Garcia de Orta 13(1):15–18.

Basu Chaudhary, K.C., and **Mathur, S.B.** 1979. Infection of sorghum seeds by *Colletotrichum graminicola*. 1. Survey, location in seed and transmission of the pathogen. Seed Science and Technology 7(1):87–92.

Beard, S.W., and **Van Wyk, P.S.** 1985. *Neocosmospora vasinfecta* pathogenic to groundnuts in South Africa. Phytophylactica 17(1):49–50.

Bhale, M.S., and **Khare, M.N.** 1982. Seed-borne fungi of sorghum in Madhya Pradesh and their significance. Indian Phytopathology 35(4):676–678.

Bhatt, R.S. 1946. Studies in Ustilaginales. I. The mode of infection of the bajra plant (*Pennisetum typhoides* Stapf.) by the smut *Tolyposporium penicillariae* Bref. Journal of the Indian Botanical Society 25(4):163–186.

Booth, C. 1970. *Fusarium oxysporum*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 211.

Booth, C. 1971. The genus *Fusarium*. Kew, Surrey, UK: Commonwealth Mycological Institute. 237 pp.

Booth, C. 1978a. *Fusarium udum*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 575.

Booth, C. 1978b. *Fusarium equiseti*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 571.

Booth, C., and **Pirozynski, K.A.** 1967. *Leptosphaerulina trifolii*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 146.

Booth, C., and **Waterson, J.M.** 1964. *Gibberella fujikuroi*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 22.

Bretag, T.W., and **Mebalds, Mc.I.** 1987. Pathogenicity of fungi isolated from *Cicer arietinum* (chickpea) grown in north-western Victoria. Australian Journal of Experimental Agriculture 27(1):141–148.

Butler, E.J. 1906. The wilt disease of pigeonpea and pepper. Agriculture Journal of India 1:25–26.

Butler, E.J. 1907. Some diseases of cereals caused by *Sclerospora graminicola*. Memoirs of the Department of Agriculture in India, Botanical Series 2:1–24.

Butler, E.J. 1910. The wilt disease of pigeonpea and the parasitism of *Neocosmospora vasinfecta* Smith. Memoirs of the Department of Agriculture in India, Botanical Series 2(9): 1–64.

Butler, E.J. 1918. Fungi and diseases in plants. Dehradun, India: Bishen Singh, Periodical Experts. 547 pp.

Cannon, P.F., and **Hawksworth, D.L.** 1984. A review of the genus *Neocosmospora* (Hypocreales). Transactions of the British Mycological Society 82(4):673–688.

Chidambaram, P., Mathur, S.B., and **Neergaard, P.** 1973. Identification of seed-borne *Drechslera* species. Friesa 10(3):165–207.

Chohan, J.S., and **Gupta, V.K.** 1968. Aflaroot, a new disease of groundnut, caused by *Aspergillus flavus* Link. Indian Journal of Agricultural Sciences 38(3):568–570.

Conci, V.C., Vásquez, A., and **None, S.F.** 1985. [Study of the form of transmission of *Fusarium oxysporum* f. sp. *ciceris* on chick pea (*Cicer arietinum* L.).] Estudio de la forma de transmisiôn de *Fusarium oxysporum* f.sp. *ciceris* en garbanzo (*Cicer arietinum* L.). (In Es. Summary in En.) Fitopatologîa 20(2):57–64.

Dalal-Clayton, D.B. 1981. Black's Agricultural dictionary. London, UK: A&C Black Publishers Ltd. 499 pp.

Das, G.N., and **Sen Gupta, P.K.** 1961. A stemphylium leaf spot disease of gram. Plant Disease Reporter 45(12):979.

Dastur, J.F. 1935. Gram wilts in the central provinces. Agriculture and Livestock in India 5(4):615–627.

Doggett, H. 1988. Sorghum diseases. Pages 342–367 *in* Sorghum. 2nd edn. New York, USA: Longman.

Duhan, J.C., and **Thakur, D.P.** 1977. Relative efficacy of fungicides in reducing pyricularia leaf spot of bajra. Haryana Agricultural University Journal of Research 7(4):185–188.

Dutta, A.K., and **Jha, D.K.** 1983. Seed-borne fungi of ragi (*Eleusine corocana* L.) in Chotanagpur and their control. Pesticides 17(4):28.

Dwivedi, R.P., and **Saksena, H.K.** 1975. Web blight disease of arhar (*Cajanas cajan* (L.) Millsp.) caused by *Thanetophorus cucumeris*. Indian Journal of Farm Sciences 3:113–114.

Ellis, M.A., and **Paschal, E.H.** 1979. Effect of fungicide seed treatment on internally seedborne fungi, germination and field emergence of pigeonpea (*Cajanus cajan*). Seed Science and Technology 7(1):75–81.

Ellis, M.B. 1966. Dematiaceous Hyphomycetes. VII. Curvularia, Brachysporium etc. Mycological Paper no. 106. Kew, Surrey, UK: Commonwealth Mycological Institute. 57 pp.

Ellis, M.B. 1971. Dematiaceous Hyphomycetes. Kew, Surrey, UK: Commonwealth Mycological Institute. 608 pp.

Ellis, M.B., and **Gibson, I.A.S.** 1975. *Cochliobolus setariae* (conidial state: *Drechslera setariae*). CMI Descriptions of Pathogenic Fungi and Bacteria no. 473.

Ellis, M.B., and **Holliday, P.** 1971a. *Trichometasphaeria turcica* (conidial state: *Drechslera turcica*). CMI Descriptions of Pathogenic Fungi and Bacteria no. 304.

Ellis, M.B., and **Holliday, P.** 1971b. *Dreschslera sacchari*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 305.

Ellis, M.B., and **Holliday, P.** 1972. *Cochliobolus nodulosus* (conidial state: *Drechslera nodulosa*). CMI Descriptions of Pathogenic Fungi and Bacteria no. 341.

Ellis, M.B., and **Waller, J.M.** 1974. *Sclerotinia fuckeliana* (conidial state: *Botrytis cineria*). CMI Descriptions of Pathogenic Fungi and Bacteria no. 431.

Ellis, M.B., and **Holliday, P.** 1976. *Drechslera sorghicola*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 491.

El-Magraby, O.M.O., and **El-Maraghy, S.S.M.** 1988. Mycoflora and mycotoxins of peanut (*Arachis hypogaea* L.) seeds in Egypt. III. Cellulose-decomposing and mycotoxin producing fungi. Mycopathologia 104(1):19–24.

El-Shafey, H.A., Abdel-Rahim, M.F., and **Refaat, M.M.** 1979. A new cephalosporium wilt of grain sorghum in Egypt. Pages 514–532 *in* Proceedings of Egyptian Phytopathological Congress 3rd. 1977, Cairo, Egypt.

Fatima, R., Mathur, S.B., and **Neergaard, P.** 1974. Importance of *Drechslera maydis* on seed crops other than maize. Seed Science and Technology 2(3):371–383.

Feakin, S.D. 1973. Pest control in groundnuts. PANS Manual no. 2. 3rd edn. London, UK: Centre for Overseas Pest Research. 197 pp.

Figueiredo, M.B., and **Cardoso Rosa, M.G.** 1968. [Occurrence of gray mould of groundnut in São Paulo state.] Ocurrencia do môfo Cinzento do Amendoim no Estado de São Paulo. (In Pt. Summary in En.) Biológico 34(10):217–220.

Francis, S.M., and **Williams, R.J.** 1983a. *Sclerospora graminicola*. CMI Descriptions of Pathogenic Fungi and Bacteria no.770.

Francis, S.M., and **Williams, R.J.** 1983b. *Peronosclerospora sorghi*. CMI Descriptions of Pathogenic Fungi and Bacteria no.761.

Frank, Z.R. 1986. New pest and disease records. Black-hull of peanut pods in Israel. Phytoparasitica 14(3):235.

Frederiksen, R.A. 1984. Acremonium wilt. Pages 49–51 *in* Sorghum root and stalk rots a critical review: Proceedings of the consultative group discussion on research needs and stratagies for control of sorghum root and stalk rot diseases, 27 Nov–2 Dec 1983, Bellagio,

Italy. Patancheru, A.P. 502 324, India: International Crops Research Institute for the Semi-Arid Tropics.

Frederiksen, R.A. (ed.) 1986. Compendium of sorghum diseases. St. Paul, Minnesota, USA: American Phytopathological Society. 82 pp.

Gams, W. 1968. Two new species of Wardomyces. Transactions of the British Mycological Society 51(5):798–802.

Garren, K.H., and **Higgins, B.B.** 1947. Fungi associated with runner peanut seeds and their relation to concealed damage. Phytopathology 37(7):512–522.

Garren, K.H., and **Wilson, C.** 1951. Peanut diseases. Pages 262-324 *in* 'The Peanut', the unpredictable legume. Washington, D.C., USA: National Fertilizer Association.

Ghewande, M.P., Pande, R.N., and **Shukla, A.K.** 1985. An out break of Fusarium wilt of groundnut in Saurashtra. Indian Botanical Reporter 4(1):61.

Ghewande, M.P., Nagpal, V., and **Reddy, P.S.** 1987. Plant protection in groundnut. Technical Bulletin. Junagarh, Gujarat, India: National Research Centre for Groundnut. 35 pp.

Ghewande Raj Jhala, M.P., Pande R.N., and **Shukla, A.K.** 1987. Leaf spots of groundnut. Indian Phytopathology 40(3):432–433.

Gilman, G.A. 1965. Black and mouldy groundnuts in the Gambia. Commonwealth Phytopathological News 4:1–2.

Grewal, J.S., and **Pal, M.** 1965. Seed mycroflora. 1. Seed-borne fungi of ragi (*Eleusine corocana* Gaertn.) their distribution and control. Indian Phytopathology 18(1):33–37.

Grewal, J.S. 1982. Control of important seed-borne pathogens of chickpea. Indian Journal of Genetics and Plant Breeding 42(3):393–398.

Grewal, J.S., and **Laha, S.K.** 1983. Chemical control of botrytis blight of chickpea. Indian Phytopathology 36(3):516–520.

Grewal, J.S., Pal, M. and **Kulshrestha, D.D.** 1974. A new record of wilt of gram caused by *Fusarium solani.* Current Science 43(23):767.

Grower, R.K., and **Suryanarayana, D.** 1970. Effect of fungicidal treatments on mycroflora of seeds of bajra during storage. Pages 547–557 *in* Plant disease problems: proceedings of the first International Symposium on Plant Pathology, 27 Dec 1966–1 Jan 1967, Pusa, New Delhi, India: Indian Agricultural Research Institute.

Haware, M.P., and **Nene, Y.L.** 1976. Some uncommon but potentially serious diseases of chickpea. Tropical Grain Legume Bulletin 5:26–30.

Haware, M.P., and **Nene, Y.L.** 1981. Phoma blight – a new disease of chickpea. Plant Disease 65(3):282.

Haware, M.P., and **Kannaiyan, J.** 1992. Seed transmission of *Fusarium udum* in pigeonpea and its control by seed-treatment fungicides. Seed Science and Technology 20(3): 597-601.

Haware, M.P., Nene, Y.L., and **Rajeswari, R.** 1978. Eradication of *Fusarium oxysporum* f.sp. *ciceri* transmitted in chickpea seed. Phytopathology 68(9):1364–1367.

Haware, M.P., Nene, Y.L., and **Mathur, S.B.** 1986. Seed-borne diseases of chickpea. Technical Bulletin no.1. Copenhagen, Denmark: Danish Government Institute of Seed Pathology for Developing Countries. 32 pp.

Hepperly, P.R., and **Rodriguez, R.** 1984. Mycofloral succession and viability losses in pigeonpea seed in Puerto Rico. Journal of Agriculture of the University of Puerto Rico 68(1):19–31.

Hiremath, P.C., and **Sulladmath, V.V.** 1985. Zonate leaf blight – a new disease of finger millet. Current Science 54(18):935.

Holliday, P. 1980. Fungus diseases of tropical crops. Cambridge, UK: Press Syndicate of University of Cambridge. 607 pp.

Holliday, P. 1989. A dictionary of plant pathology. Cambridge, UK: Press Syndicate of the University of Cambridge. 369 pp.

Holliday, P., and **Punithalingam, E.** 1970. *Macrophomina phaseolina*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 275.

India : Ministry of Agriculture and Irrigation. 1977. Proceedings of the Meetings of the Indian Council of Agricultural Research Committee on Quarantine Arrangements for Import of Pearl Millet Seeds by International Crops Research Institute for the Semi-Arid Tropics. CPPTI, Hyderabad, India, Hyderabad, A.P. India: Ministry of Agriculture and Irrigation. 2 pp. (Limited distribution).

Jackson, C.R. 1962. Aspergillus crown rot of peanut in Georgia. Plant Disease Reporter 46(12):888–892.

Jackson, C.R., and **Bell, D.K.** 1969. Diseases of peanut (groundnut) caused by fungi. Research Bulletin, University of Georgia, Experiment Stations no. 56. 137 pp.

Joffe, A.Z., and **Borut, S.Y.** 1966. Soil and kernal mycoflora of groundnut fields in Israel. Mycologia 58(4):629–640.

Joshi, M.M., and **Singh, R.S.** 1969. A botrytis grey mold of gram. Indian Phytopathology 22(1):125–128.

Kaiser, W.J. 1972. Occurrence of three fungal diseases of chickpea in Iran. FAO Plant Protection Bulletin 20(4):73–78.

Kamal, and **Verma, A.K.** 1979. Seed-borne mycoflora of arhar (T-21), effect of culture filtrates of some isolates on seed germination and fungicidal treatments. Indian Journal of Mycology and Plant Pathology 9(1):41–45.

Kannaiyan, J., and **Nene, Y.L.** 1977. Alternaria leaf spot of pigeonpea. Tropical Grain Legume Bulletin 9:34.

Kannaiyan, J., Nene, Y.L., and **Sheila, V.K.** 1980. Control of mycoflora associated with pigeonpea seeds. Indian Journal of Plant Protection 8(2):93–98.

Kavita Rani, Madan Mohan, and **Mukerji, K.G.** 1978. Studies on seed-borne fungi. I. Occurrence of three pathogenic fungi on sorghum seeds. Seed Research 6(1):38–42.

Kenneth, R. 1975. *Sclerospora graminicola.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 452.

Khanna, A., and **Payak, M.M.** 1971. Electron microscopy of teliospores of *Melanopsichum eleusinis* (ragi smut). Current Science 40(19):529.

Khune, N.N., and **Kapoor, J.N.** 1981. A new disease of pigeonpea. Indian Phytopathology 34(2):258–260.

Kocatürk, S., and **Maden, S.** 1977. [Studies on the protective action of certain chemicals against seedborne fungus diseases of groundnut.] yerfistiginda tohumla tasinan hastalik etmeni funguslara karşi muhtelif ilâçlarin koruyucu etkileri Üzerinde calismalar. (In Tr. Summary in En.) Pages 95–96 *in* Zarai Mücadele Araştirma yilligi no. 11. Ankara, Turkey: Regional Plant Protection Research Institute.

Kolte, S.J. 1984. Diseases of annual edible oilseed crops. vol. 1. Peanut diseases. Boca Raton, Florida, USA: CRC Press. 143 pp.

Konde, B.K., Dhage, B.V., and **More, B.B.** 1980. Seedborne fungi of some pearl millet cultivars. Seed Research 8(1):59–63.

Kore, S.S., and **Bhide, V.P.** 1975. Resistance in some sorghum cultivars to *Curvularia* spp. in Maharashtra State. Sorghum Newsletter 18:46–47.

Kovachevsky, I.C. 1936. [The blight of chickpea, *Mycosphaerella rabiei* n. sp.] (In Ru.) Sofia, Bulgaria: Ministry of Agriculture Domains. 80 pp.

Kraft, J.M. 1969. Chickpea, a new host of *Fusarium solani* f. sp. *pisi.* Plant Disease Reporter 53(2):110–111.

Kranz, J., and **Pucci, E.** 1963. Studies on soil-borne rots of groundnuts (*Arachis hypogaea* L.). Phytopathologische Zeitschrift 47(2):101–112.

Krishna Prasad, N.V., and **Basuchaudhary, K.C.** 1987. Seed-borne mycoflora of ragi (*Eleusine corocana* (L.) Gaertn.) from Andhra Pradesh and their control. International Journal of Tropical Plant Diseases 5(2):181–187.

Kulkarni, B.G.P., Seshadri, V.S., and **Hegde, R.K.** 1976. The perfect stage of *Sphacelia sorghi* McRae. Mysore Journal of Agricultural Sciences 10(2):286–289.

Kulkarni, G.S. 1913. Observations on the downy mildew of bajra and jowar. Memoirs of the Department of Agriculture in India, Botanical Series 5(5):268–273.

Kulkarni, G.S. 1922. The smut of nachani or ragi (*Eleusine corocana* Gaertn.). Annals of Applied Biology 9(3):184–186.

Kumar, A., and **Viswanath.** 1991. Fungicidal control and disease rating-scale of long smut (*Tolyposporium ehrenbergii*) of sorghum (*Sorghum bicolor*). Indian Journal of Agricultural Sciences 61(3):225–227.

Kumar, K. 1980. Seed-borne nature of *Phyllosticta cajani* in pigeonpea, *Cajanus cajan*. Page 61 *in* Summaries of research projects 1967–88 (Mathur, S.B., ed.). Hellerup, Denmark: Danish Institute of Seed Pathology for Developing Countries.

Labrousse, F. 1931. [Anthracnose of chickpea.] L'anthracnose du pois-chiche. (In Fr. Summary in En.) Revue de Pathologie Vegetale et d' Entomologie Agricole de France 18(6):226–231.

Labuschagne, N., Kotze, J.M., and **Wehner, F.C.** 1980. *Thielaviopsis basicola* infection of groundnuts in South Africa. Phytophylactica 12(3):177–180.

Laha, S.K., and **Grewal, J.S.** 1983. Botrytis blight of chickpea and its perpetuation through seed. Indian Phytopathology 36(4):630–634.

Leukel, R.W., and **Johnson, A.G.** 1948. *Periconia circinata,* the cause of milo disease. Science 107(2769):93–94.

Leukel, R.W., Martin, J.H., and **Lefebvre, C.L.** 1951. Sorghum diseases and their control. Farm Bulletin no. 1959. U.S. Department of Agriculture.

Lewin, H.D., and **Natarajan, S.** 1971. Control of dry root rot *Rhizoctonia bataticola* (Taub.) Butl. in groundnut by seed treatment. Madras Agricultural Journal 58(5):395–404.

Lima, D.M.M., and **Aguillar, J.A.E.** 1978. [Groundnut wilt caused by *Fusarium solani* (Mart.) Sacc.] Murcha-do-amendoim Causada por *Fusarium solani* (Mart.) Sacc. (In Pt. Summary in En.) Pesquisa Agropecuária Brasileira 13(3):1–5.

Lokesh, M.S., Hiremath, R.V., and **Hegde, R.K.** 1987. Seed mycoflora of red gram [*Cajanus cajan* (L.) Millsp.] Plant Pathology Newsletter 5(1–2):31.

Lo'pez Rosa, J.H. 1969. *Phoma* sp, the causal agent of pigeonpea canker. Phytopathology 59(10):1348. (Abstract.)

Maden, S. 1987. Seed borne fungal diseases of chick-pea in Turkey. Journal of Turkish Phytopathology 16(1):1–8.

Maden, S., Singh, D., Mathur, S.B., and **Neergaard, P.** 1975. Detection and location of seed-borne inoculum of *Ascochyta rabiei* and its transmission in chickpea (*Cicer arietinum*). Seed Science and Technology 3(3–4):667–681.

Manzo, S.K. 1976. Studies on the mode of infection of sorghum by *Tolyposporium ehrenbergii*, the causal organism of long smut. Plant Disease Reporter 60(11):948–952.

Mathur, K., Siradhana, B.S., and **Lodha, B.C.** 1987. Studies on seedling blight of sorghum caused by *Gloeocercospora sorghi*. Seed Science and Technology 15(3):851–858.

Mathur, R.S., and **Jai Prakash** 1976. New records of fungi isolated from seed and leaf spots of some pasture plants in India. Indian Phytopathology 29(1):54–55.

Mathur, S.K., Mathur, S.B., and **Neergaard, P.** 1975. Detection of seed-borne fungi in sorghum and location of *Fusarium moniliforme* in the seed. Seed Science and Technology 3(3–4):683–690.

Mercer, P.C., and **Kisyombe, C.T.** 1978. The fungal flora of groundnut kernels in Malawi and the effect of seed-dressing. Pest Articles and News Summary (PANS) 24(1):35–42.

Mishra, A., and **Siradhana, B.S.** 1978. Chemical control of anthracnose of sorghum. Indian Phytopathology 31(2):225–227.

Mishra, R.P., Sharma, N.D., and **Joshi, L.K.** 1975. A new disease of gram (*Cicer arietenum* L.) in India. Current Science 44(17):621–622.

Misra, A.P. 1959. Diseases of millets and maize. A review of work done in India during 1931–1958. Indian Agriculturist 3(2):75–89.

Misra, A.P., Om Prakash, and **Mishra, B.** 1974. An eye spot disease of bajra caused by *Helminthosporium sacchari* from India. Indian Phytopathology 27(1):101–102.

Mitra, M. 1921. Morphology and parasitism of *Acrothecium pennisetum* n. sp. (A new disease of *Pennisetum typhoideum*). Memoirs of the Department of Agriculture in India, Botanical Series 11(3):57–74.

Mitra, M. 1923. *Helminthosporium* spp. on cereals and sugarcane in India. Part I. Diseases of *Zea mays* and *Sorghum vulgare* caused by species of *Helminthosporium*. Memoirs of the Department of Agriculture in India, Botanical Series 11(10):219–242.

Mitra, M., and **Mehta, P.R.** 1934. Diseases of *Eleusine corocana* Gaertn., and *E. aegyptaica* Desf. caused by species of *Helminthosporium*. Indian Journal of Agricultural Sciences 4(6):943–975.

Mordue, J.E.M. 1967. *Colletotrichum graminicola*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 132.

Mordue, J.E.M. 1974a. *Thanetophorus cucumeris*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 406.

Mordue, J.E.M. 1974b. *Corticium rolfsii*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 410.

Mridha, A.U., and **Fakir, G.A.** 1978. Seed transmission of *Macrophomina phaseolina* and *Sclerotium rolfsii* in groundnut (*Arachis hypogeae* L.) in Bangladesh. Bangladesh Journal of Botany 7(2):31–34.

Mulder, J.L., and **Holliday, P.** 1971. *Gloeocercospora sorghi.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 300.

Mulder, J.L., and **Holliday, P.** 1974. *Cercospora sorghi.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 419.

Mundkur, B.B., and **Thirumalachar, M.J.** 1952. Ustilaginales of India. Kew, Surrey, UK: Commonwealth Mycological Institute. 84 pp.

Nayudu, M.V. 1963. *Leptosphaerulina arachidicola* on groundnut. Indian Phytopathology 16(4):384–386.

Neergaard, P. 1979. Seed pathology. vol 1. Rev. edn. London, UK: Macmillan Press. 839 pp.

Nelson, P.E., Toussoun, T.A., and **Marasas, W.F.O.** 1983. Fusarium species: an illustrated manual for identification. Pennsylvania, USA: Pennsylvania State University Press. 193 pp.

Nene, Y.L. 1980. Diseases of chickpea. Pages 171–178 *in* Proceedings of the International Workshop on Chickpea Improvement, 28 Feb–2 Mar 1979, Hyderabad, India. Patancheru, A.P. 502 324, India: International Crops Research Institute for the Semi-Arid Tropics.

Nene, Y.L., and **Singh, S.D.** 1976. Downy mildew and ergot of pearl millet. Pest Articles and News Summary (PANS) 22(3):366–385.

Nene, Y.L., and **Reddy, M.V.** 1987. Chickpea diseases and their control. Pages 233–270 *in* The chickpea (Saxena, M.C., and Singh, K.B., eds.). Wallingford, Oxon, UK: CAB International.

Nene, Y.L., Sheila, V.K., and **Sharma, S.B.** 1989. A world list of chickpea (*Cicer arietinum* L) and pigeonpea [*Cajanus cajan* (L.) Millsp.] pathogens. Legumes Pathology Progress Report 7. Patancheru, A.P. 502 324, India: Legumes Program, International Crops Research Institute for the Semi-Arid Tropics. 23 pp. (Limited distribution.)

Olive, L.S., Lefebvre, C.L., and **Sherwin, H.S.** 1946. The fungus that causes sooty stripe of sorghum spp. Phytopathology 36(3):190–200.

Onions, A.H.S. 1966a. *Aspergillus flavus.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 91.

Onions, A.H.S. 1966b. *Aspergillus niger.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 94.

Orian, G. 1949. Division of Plant Pathology. Report, Department of Agriculture, Mauritius 1948:69–74.

Peregrine, W.T.Z. 1971. Groundnut rust (*Puccinia arachidis*) in Brunei. Pest Articles and News Summary (PANS) 17(3):318–319.

Phelps, R.H., Oudit, I.L., and **Page, C.** 1974. Southern blight (*Sclerotium rolfsii*), a new disease on pigeonpea (*Cajanus cajan*). Pages 63–70 *in* Proceedings, Symposium on the Protection of Horticultural Crops in the Caribbean. St. Augustine, Trinidad, West Indies. St Augustine, Trinidad, West Indies: University of the West Indies, Department of Crop Science.

Porter, D.M., and **Beute, M.K.** 1974. Sclerotinia blight of peanuts. Phytopathology 64(2):263–264.

Porter, D.M., and **Mozingo, R.W.** 1986. Importance of seed transmission in the spread of *Cylindrocladium crotalariae*. Peanut Science 3(2):80–82.

Porter, D.M., Smith, D.H., and **Kabana, R.R.** (eds.). 1984. Compendium of peanut diseases. St. Paul, Minnesota, USA: American Phytopathological Society. 73 pp.

Prasada, R., and **Goyal, J.P.** 1970. A new species of *Pyricularia* on bajra. Current Science 39(12):287–288.

Punithalingam, E. 1976. *Botryodiplodia theobromae.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 519.

Punithalingam, E. 1985. *Phoma sorghina*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 825.

Punithalingam, E., and **Gibson, I.A.S.** 1976. *Phoma medicagenis* var. *pinodella*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 518.

Punithalingam, E., and **Holliday, P.** 1972a. *Mycosphaerella pinodes*. CMI Descriptions of Pathogenic Fungi and Bacteria no. 340.

Punithalingam, E., and **Holliday, P.** 1972b. *Ascochyta rabiei.* CMI Descriptions of Pathogenic Fungi and Bacteria no. 337.

Rachie, K.O., and **Peters, L.V.** 1977. Plant protection. Pages 86–102 *in* The Eleusines: a review of the world literature. Patancheru, A.P. 502 324, India: International Crops Research Institute for the Semi-Arid Tropics.

Rachie, K.O., and **Majumdar, J.V.** 1980. Growing the crop. Pages 127–191 *in* Pearl millet. Pennsylvania, USA: Pennsylvania State University Press.

Rajashekar, K., Shetty, H.S., and **Majumdar, S.K.** 1989. Relative efficacy of some fungicides as seed dressing agents against seed mycoflora of ragi. Pesticides 23(3):23–25.

Ramakrishnan, T.S. 1941. Top rot ('twisted top' or 'pokka bong') of sugarcane, sorghum and cumbu. Current Science 10(9):406–408.

Ramakrishnan, T.S. 1963. Diseases of millets. New Delhi, India: Indian Council of Agricultural Research. 152 pp.

Ram Nath, Neergaard, P., and **Mathur, S.B.** 1970. Identification of Fusarium species on seeds as they occur in blotter test. Proceedings of the International Seed Testing Association 35(1):121–144.

Ranganathiah, K.G., and **Rao, A.N.S.** 1982. Seed treatment of finger millet against Helminthosporiose and blast disease. Indian Journal of Mycology and Plant Pathology 12(3):319–320.

Rao, B.M., Raveesha, K.A., Prakash, H.S., and **Shetty, H.S.** 1984. Effect of chemical treatment on germination and mycoflora of pearl millet seed. Seed Research 12(2):102–103.

Raper, K.B., Fennell, D.I., and **Austwick, P.K.C.** 1965. The genus Aspergillus. Malabar, Florida, USA: Robert E. Krieger Publishing Co. 686 pp.

Rathi, Y.P.S., and **Lal, S.** 1977. A wilt disease of pigeonpea caused by *Macrophomina phaseoli* [*M. phaseolina*]. Acta Botanica Indica 5(1):83–84.

Ravinder Reddy, Ahmed, K.M., Joshi, N.C., and **Ratna, A.S.** 1990. Health status of seeds of ICRISAT mandate crops in relation to plant quarantine. Pages 210–220 *in* Proceedings of the National Seminar on Advances in Seed Science and Technology, 14–16 Dec 1989, Mysore, India. Mysore, Karnataka, India: University of Mysore, Department of Studies in Applied Botany.

Reddy, M.V., and **Kababeh, S.** 1984. Eradication of *Ascochyta rabiei* from chickpea seed with thiabendazole. International Chickpea Newsletter 10:17–18.

Reddy, M.V., Sharma, S.B., and **Nene, Y.L.** 1990. Pigeonpea: disease management. Pages 303–347 *in* The pigeonpea (Nene, Y.L., Hall, S.D., and Sheila, V.K., eds.). Wallingford, Oxon, UK: CAB International.

Reddy, M.V., Singh, K.B., and **Nene, Y.L.** 1982. Further studies of Calixin-M in the control of seed-borne infection of ascochyta blight in chickpea. International Chickpea Newsletter 6:18–19.

Safeeulla, K.M. 1976. Biology and control of the downy mildews of pearl millet, sorghum, and finger millet. Mysore, Karnataka, India: University of Mysore. 304 pp.

Saksena, H.K., Singh, G.P., and **Nath , S.** 1967. Blight disease of groundnut in India. Indian Phytopathology 20(1):67–69.

Shekhawat, K.S., Verma, C.P., and **Pathak, V.N.** 1986. Effect of seed-dressing fungicides on collar rot of groundnut caused by *Aspergillus niger*. Summa Phytopathologica 12(3–4):207–216.

Shetty, H.S., Neergaard, P., and **Mathur, S.B.** 1978. Demonstration of seed transmission of downy mildew or green ear disease, *Sclerospora graminicola*, in pearl millet, *Pennisetum typhoides*. Proceedings of the Indian National Science Academy, B 43(6):201–206.

Shetty, H.S., Mathur, S.B., and **Neergaard, P.** 1980. *Sclerospora graminicola* in pearl millet seeds and its transmission. Transactions of the British Mycological Society 74(1):127–134.

Shetty, H.S., Mathur, S.B., Neergaard, P., and **Safeeulla, K.M.** 1982. *Drechslera setariae* in Indian pearl millet seeds, its seed-borne nature, transmission and significance. Transactions of the British Mycological Society 78(1):170–173.

Shetty, H.S., Gopinath, A., and **Rajashekar, K.** 1985. Relationship of seed-borne inoculum of *Pyricularia grisea* to the incidence of blast of finger millet in the field. Indian Phytopathology 38(1):154–156.

Shree, M.P. 1983. Efficacy of a few fungicides against the incidence of leaf blight of jowar. Pesticides 17(10):27–29.

Singh, D.S., and **Pavgi, M.S.** 1977. Perpetuation of *Pyricularia penniseti* causing brown leaf spot of bajra. Indian Phytopathology 30(2):242–244.

Singh, H.B. 1988. Leaf blight of pigeonpea by *Fusarium semitectum.* International Pigeonpea Newsletter 7:29–30.

Singh, I., and **Chohan, J.S.** 1976. Fungi associated with seeds of gram (*Cicer arietinum*) and control of pathogenic ones. Indian Journal of Myclogy and Plant Pathology 6(1):71–72.

Sivanesan, A. 1987. Graminicolous species of Bipolaris, Curvularia, Drechslera, Exserohilum and their teleomorphs. Mycological Papers no. 158. Kew, Surrey, UK: Commonwealth Mycological Institute. 261 pp.

Smith, D.H. 1984. Pepper spot and leaf scorch. Pages 10–11 *in* Compendium of peanut diseases (Porter, D.M., Smith, D.H., and Rodriguez-Kabana, R., eds.). St. Paul, Minnesota, USA: American Phytopathological Society.

Snell, W.H., and **Dick, E.A.** 1971. A glossary of mycology. Cambridge, USA: Harvard University Press. 181 pp.

Sprague, R. 1950. Diseases of cereals and grasses in North America. New York, USA: Ronald Press. 538 pp.

Subramanian, C.V. 1971. Hyphomycetes. New Delhi, India: Indian Council of Agricultural Research. 930 pp.

Subrahmanyam, P., and **McDonald, D.** 1982. Groundnut rust — its survival and carry-over in India. Proceedings of the Indian Academy of Sciences (Plant Sciences) 91(2):93–100.

Subrahmanyam, P., McDonald, D., Siddaramaiah, A.L., and **Hegde, R.K.** 1981. Leaf spot and veinal necrosis disease of groundnut in India caused by *Alternaria alternata.* FAO Plant Protection Bulletin 29(3–4):74–76.

Suhag, L.S. 1973. Mycoflora of gram (*Cicer arietinum*) seeds, pathology and control. Indian Journal of Mycology and Plant Pathology 3(1):40–43.

Sutton, B.C. 1980. The Coelomycetes, fungi imperfecti with pycnidia, acervuli, and stroma. Kew, Surrey, UK: Commonwealth Mycological Institute. 696 pp.

Swarup, G., Hansing, E.D., and **Rogerson, C.T.** 1962. Fungi associated with sorghum seed in Kansas. Transactions of the Kansas Academy of Science 65(2):120–137.

Tarr, S.A.J. 1962. Diseases of sorghum, sudan grass and broom corn. Kew, Surrey, UK: Commonwealth Mycological Institute. 380 pp.

Thakur, D.P. 1984. Ergot disease of pearl millet. Review of Tropical Plant Pathology 1:297–328.

Thakur, R.P., and **King, S.B.** 1988. Smut disease of pearl millet. Information Bulletin no. 25. Patancheru, A.P. 502 324, India: International Crops Research Institute for the Semi-Arid Tropics. 20 pp.

Thakur, R.P., Rao, V.P., and **Williams, R.J.** 1984. The morphology and disease cycle of ergot caused by *Claviceps fusiformis* in pearl millet. Phytopathology 74(2):201–205.

Thirumalachar, M.J., Shaw, C.G., and **Narasimhan, M.J.** 1953. The sporangial phase of the downy mildew on *Eleusine corocana*, with a discussion on the identity of *Sclerospora macrospora* Sacc. Bulletin of the Torrey Botanical Club 80:299–307.

Udagawa, S.I. 1963. Neocosmospora in Japan. Transactions of the Mycological Society of Japan 4(5):121–125.

Uppal, B.N., Kolhatkar, K.G., and **Patel, M.K.** 1936. Blight and hollow stem of sorghum. Indian Journal of Agriculture Science 6(6):1323–1334.

Van Hoof, H.A. 1949. Infection of *Sorghum vulgare* Pers. by *Colletotrichum falcatum* Went. Landbouw 21(6):267–276.

Venkatarayan, S.V. 1946. Diseases of ragi (*Eleusine corocana*). Mysore Agricultural Journal 24(2):50–57.

Venkateswarulu, S., Reddy, A.R., Singh, O.N., and **Chauhan, V.B.** 1981. Alternaria blight: the most serious disease of post-rainy season (rabi) pigeonpeas. International Pigeonpea Newsletter 1:28–29.

Vidyasekaran, P. 1983. Control of *Fusarium moniliformae* infection in sorghum seed. Seed Science and Technology 11(2):435–439.

Vidyasekaran, P., Thulasidas, G., Ramasamy, K.R., and **Kandaswamy, T.K.** 1980. Preservation of viability of sorghum seeds by controlling seed-borne fungi. Indian Phytopathology 33(2):225–230.

Vishwakarma, S.N., and **Chaudhry, K.C.B.** 1974. Floral blight of gram incited by *Alternaria alternata*. Netherlands Journal of Plant Pathology 80(3):110–112.

Vishwakarma, S.N., and **Basu Chaudhary, K.C.** 1982. Seed treatment to control root diseases of gram. Pesticides 16(11):33–34.

Wadsworth, D.F., and **Melouk, H.A.** 1985. Potential for transmission and spread of *Sclerotinia minor* by infected peanut seed and debris. Plant Disease 69(5):379–381.

Wallace, G.B. 1950. Report of the Plant Pathologist. Annual Report, Department of Agriculture, Tanganyika 1948:145–147.

Wallace, G.B., and **Wallace, M.M.** 1953. Diseases of sorghum. Tanganyika Department of Agriculture, Pamphlet, no. 53.

Watanabe, T., and **Hashimoto, K.** 1978. Recovery of *Gloeocercospora sorghi* from sorghum seed and soil, and its significance in transmission. Annals of the Phytopathological Society of Japan 44(5):633–640.

Weimer, J.L., Burton, G.W., and **Higgins, B.B.** 1937. *Ascochyta sorghina* Sacc. on sudan grass, johnson grass, and sorghums in Georgia. Plant Disease Reporter 21(21):378.

Wells, H.D. 1967. Effects of temperature on pathogenicity of *Helminthosporium setariae* on seedlings of pearl millet, *Pennisetum typhoides*. Phytopathology 57(9):1002.

Wells, H.D., and **Winstead, E.E.** 1965. Seed-borne fungi in Georgia-grown and Western-grown pearl millet seed on sale in Georgia during 1960. Plant Disease Reporter 49(6):487–489.

Williams, R.J., and **Singh, S.D.** 1981. Control of pearl millet downy mildew by seed treatment with metalaxyl. Annals of Applied Biology 97(3):263–268.

Wu, W.S. 1979. Botrytis blight on peanut. [*B. cinerea* on groundnut]. NTU (National Taiwan University) Phytopathologist and Entomologist 6:58–60.

Wu, W.S. 1983. Sorghum diseases in Taiwan and characterization and control of its seed-borne pathogens. Plant Protection Bulletin, Taiwan 25(1):1–13.

Glossary

g kg^{-1} grams per kilograms.
mm millimeter (1 mm = 0.10 cm).
μm micrometer (1 μm = 10^{-6} m, previously micron: μ).
UV ultraviolet radiation (wave length 40–400 nm).
NUV near ultraviolet radiation (wave length 320–420 nm).
Acervulus (pl=acervuli) a compact subepidermal fruiting body consisting of mat of hyphae giving rise to short conidiophores, conidia, and sometimes setae on the upper surface.
Acicular (of a spore) which is needle-shaped, slender, and pointed.
Acrogenous conidia borne at the apex of the conidiophore.
Acropleurogenous conidia borne at the apex and on the sides of the conidiophore.
Acropetal conidia produced successively towards the apex, having the youngest conidium at the apex.
Aerial mycelium mycelium which grows above the substrate frequenting the air.
Aflaroot a disease of groundnuts caused by the fungus *Aspergillus flavus*.
Aggregate crowded together.
Amphigenous growth of the fungus all around or on two sides.
Amyloid (of spore walls, spore ornamentation, hyphal walls, ascus tips, etc) stained blue-black in Melzer's reagent, presumably because of the presence of starch or starch-like compound.
Ampulliform flask shaped.
Angular (of spores) not regular in outline; not rounded.
Anthracnose a plant disease having characteristic limited lesions, necrosis, and hypoplasia caused by acervular fungi.
Apical (of spore) referring to the apex; situated at the end.
Apiculate refers to a hilar appendage of the conidium having a short projection at one end.
Apically flattened (of conidia or fruiting bodies) flattened at the apex.
Appendage usually referring to a filiform outgrowth of a conidium.
Ascospore a spore produced in an ascus.
Ascus (pl=asci) a sac-like reproductive cell of the perfect stage of the Ascomycetes, in which ascospores are produced.
Aseptate a cell without cross walls or septa.
Basitonous conidiophore arising near the base of a hypha.
Beak (of pycnidia or perithecia) an elongated neck through which spores are discharged; hook-like projection at one end or an extended apical cell in the case of a conidium.
Bicelled having two cells.
Biguttulate (of spores) having two spherical oil droplets within each cell.
Biseriate (of spores or fruiting bodies) arrangement in two rows or series.
Bitunicate (of asci) having two walls, the outer of which is inelastic and ruptures at maturity, releasing an extensible inner wall that stretches until it protrudes from the ostiole before the spore is discharged.
Blight a kind of plant disease which is characterized by extensive spotting, discoloration or destruction of leaves, flowers, stems or entire plant, and is often coupled with the name of the affected part.
Blotter test a method of seed health inspection in which seeds are incubated on moistened blotter papers to test the presence of microorganisms.
Cancerous cancer-like.
Catanulate or **Catenate** (of spores) developing in chains; linked as in a chain.
Cell basic unit of structure and function of all living things consisting of a mass of protoplasm and nucleus.

Chlamydospore a thick-walled, nonsexual (asexual) resting spore developed by thickening of hyphal cell or cells. Intercalary: When chlamydospores are formed between the apex and the basal cells. Terminal: when they are formed at the apex.

Cirrhus (or **Cirrus)** (pl = cirri or cirrhi) mass of spores laden with mucus forcing out of an ostiole in a ribbon-like fashion.

Clavate (of fructifications, spores, etc.) club-shaped, which is narrowing towards the base, or thickened towards the apex than the base.

Coalesce (of fructifications) running together; joining together.

Collar rot rotting of the stem near the soil surface due to infection by a pathogen.

Colony a group of hyphae or a mycelium belonging to one species, growing together.

Columella (pl = columelae) a persistent, sterile, axial body within a sporangium or fruit body.

Columnar having the form of a column.

Compound microscope an optical instrument to magnify objects invisible to the naked eye. It consists of two lenses of different focal lengths mounted on the same viewer.

Concentric one circle within another with a common center.

Confluent fungal structures joined together by hyphae, cells or stromatic tissue.

Conical (of spores) more or less cone-shaped.

Conidium (pl=conidia) an asexual spore of a fungus.

Conidiophore A specialized hypha which bears a conidium. Primary conidiophore — one arising from the stroma, substrate, or hyphae. Secondary conidiophore — one borne on the conidium. Simple conidiophore — an unbranched conidiophore.

Contamination (of seed) intermixed with a pathogen; (of spores) not pure.

Cruciferae a family of dicotyledonous plants.

Cruciferous like cruciferae.

Cuticle the outer layer (of basidiocarps or fruiting bodies) consisting of compressed hyphae parallel to the surface.

Cylindrical (of spores, fructifications, etc.) round in cross section and of the same diameter throughout its length.

Cymoid (cymose) primary and secondary branches of a conidiophore terminating in conidia.

Dehisce (of fructifications) opens when mature, by a pore or slit, or by tearing.

Denticule small tooth-like projections on which conidia are borne.

Dichotomous branching, frequently successively, into two more or less equal arms.

Disease any disturbance of a plant that interferes with its normal structure, function, or economic value.

Disseminate transport of fungal inoculum.

Distoseptate of a conidium having individual cells, each surrounded by a sac-like wall distinct from the outer wall.

Doliiform barrel-shaped, cask-shaped, or jar-shaped.

Dorsal back of an organ as to the under surface of a leaf.

Dormant mycelium a thallus or a kind of mycelium, usually thick-walled resting for some time and capable of becoming active during favorable conditions.

Downy mildew a type of plant disease caused by the fungi belonging to the *Peronosporaceae.*

Echinulate conidium having pointed spines projecting from cell walls.

Ellipsoidal (of spores) elliptical in optical section.

Elliptical (of spores) having the shape of an ellipse, i.e., of foreshortened circle; an oval curve.

Embedded fixed in the substratum; surrounded by other matter.
Endemic (of a plant disease) restricted to one country or geographical region; (of fungi) native to certain region.
Endogenous (of spores) developing or originating inside the conidiogenous cell.
Endosporium the innermost membrane of the oospore which is usually thin.
Epidermis the outermost limiting layer of the thallus or the outer layer of cell in plants.
Epispore outer spore wall which determines the shape of the spore.
Eradicate to control a plant disease by eliminating the pathogen after it is established.
Ergot sclerotium of ergot fungi; the disease of cereals caused by the fungi belonging to genus *Claviceps*.
Erumpent bursting through the surface of substratum.
Externally seedborne (of fungi) located outside functional part of the seed; (of disease) the infection is present on the surface of the seeds but may be externally difficult to eradicate by ordinary seed treatments.
Exosporium the layer of the oospore that covers the 'epispore' and becomes the outermost layer after the 'perispore' disappears.
Exudate liquid discharge from diseased tissue, or in case of a fungal body not usually through an aperture as in root or leaf exudate.
Eye spot of fungal disease of cereals (*Bipolaris sacchari*) causing oval or eye-shaped areas of black dots on the leaves.
Falcate (of spores) curved like a sickle.
False head a mass of conidia formed at the tip of a conidiophore or phialide appearing like a ball without a distinct wall. The conidia are enclosed in a gelatinous matrix.
Filiform (of spores) slender and thread-like.
Flexuous easily bent alternately in opposite directions, or having turns or windings.
Floccose cotton-like groups or tufts.
Fluorescence property of emitting light in the presence of certain rays such as during absorption of ultraviolet radiation.
Floret one of the small flowers in a clustered or compact inflorescence.
Fluffy downy.
Foot rot rot involving the lower part of the stem-root axis, synonymous to collar-rot.
Foot-shape refers to the shape of the basal cell of a macroconidium produced by fungi of the genus *Fusarium* which is shaped like a foot.
Fructification a fungal structure which bears spores; spore-bearing organs like apothecium, ascocarp, basidiocarp, perithecium, pycnidium, etc.
Fungicide a chemical agent or a substance that can kill or inhibit fungi.
Fungus (pl=fungi) a group of saprophytic and parasitic lower plants that lack chlorophyll, as molds, mildews, smuts, and rusts.
Fusiform spindle-like; narrow towards the ends.
Fusoid spindle-shaped; narrowing towards the ends.
Gelatinous rubbery; jelly-like.
Geniculate bent like a knee.
Genus (pl=genera) a group of related species.
Germplasm seeds and plants used for breeding.
Germinate to begin to grow.
Germination active growth from the embryo of a seed; or a process of growth from a spore.
Globose nearly spherical.
Glume one of a pair of small scale-like bracts enclosing a grass spikelet.

Grain mold an obvious mycelial or spore mass on the grains.
Granular covered with a granule-like substance.
Gregarious (of fructifications) formed in crowded groups or close groups, but not fused or joined together.
Gray mold a type of plant disease caused by fungi belonging to the genus *Botrytis*.
Guttulate refers to the presence of oil droplets within a cell.
Guttation having tear-like drops; marked as if by drops of liquid.
Helicoid coil-like; spiral-like.
Hilum a scar or mark in the basal cell of a conidium, indicating the point of attachment to a conidiophore.
Host a living organism harboring a parasite.
Hülle cell encasing cell of vesicular nature surrounding the cleistothecium in the fungi belonging to the genus *Aspergillus*.
Humid a geographical area where the water vapor content of the air is high.
Hyaline colorless; transparent.
Hypertrophy abnormal enlargement of an organ.
Hypha (pl=hyphae) one of the filaments of a mycelium; the basic structural unit of a fungus. A hypha without septa is aseptate or coenocytic, one with septa is septate.
Immersed see 'embedded'.
Imperfect state a phase in the life cycle of a fungus in which asexual spores or no spores are formed.
Infection process of beginning or producing disease.
Inflorescence flower or flower cluster.
Incubate (of seed health testing) maintaining seeds in an environment favorable to the development of symptoms or structures of pathogens.
Inoculum infective material, or pathogens, or parts of them.
Internally seedborne (of fungus) when it is located inside the functional part of the seed; (of disease) when the infection is deep-seated. The pathogen is carried within the seed and when the seed is sown at least some of the infected seeds will give rise to diseased seedlings. The disease is directly transmitted from one generation to the next via the seed.
Lateral attached to one side.
Leaf spot circular spots on the leaves due to infection caused by pathogens.
Lesion a localized or limited diseased area.
Long smut a type of smut disease in which the entire grain is converted into compacted mass of spores protected by a persistent grain membrane.
Loose smut a type of smut disease in which the entire grain is converted into a mass of loose, unprotected spores.
Macroconidium a long or large conidium of fungi of the genus *Fusarium.*
Macroscopic large enough to be visible without the aid of the microscope.
Mesosporium a delicate layer of the oospore wall often difficult to distinguish.
Microscopic requiring the use of a microscope to be seen clearly.
Microconidium a conidium, much smaller than macroconidium, usually single-celled.
Monophyalidic phialide with single opening.
Morphology a branch of science which deals with form and structure of organisms.
Mold a microfungus having a well-marked mycelium or spore mass.
Mucoid like mucus or slime.
Multiseptate (of spores) having a number of septa.
Multiostiolate (of fructifications) having more than one ostiole.

Mummified decomposed, dried or distorted in shape.
Muriform conidium with transverse and longitudinal septa.
Mycelium a group of hyphae constituting the vegetative body of a fungus.
Navicular boat-shaped.
Nonamyloid (of spore coverings) remaining hyaline or becoming yellowish in Melzer's reagent (see amyloid).
Obclavate inverted club-shaped widest near the base (of spores, fructifications, etc.) inversely clavate.
Obclavate cylindric shape of a spore or fruiting body of the fungus which is widest near the base and is round in cross section.
Obconical conidium shaped like a cone with broad end at the base.
Obconically truncate a conical spore or fruiting body of a fungus with its terminal portion ending abruptly.
Obligate parasite a parasitic fungus that can develop only in living tissues.
Oblique septa (of spores) septa formed somewhat perpendicular to the horizontal septum as in *Alternaria*.
Oblong (of spores, fructifications, etc.) twice as long as wide and having somewhat truncate ends.
Obovoid (of spores, fructifications, etc.) reversely ovate; shaped like an inverted egg, with the narrow end at the base.
Obpyriform pear-shaped with the broad end at the base.
Obtuse rounded or blunt.
Oospore a thick-walled resting spore produced by the union of an oogonium and an antheridium.
Ooze movement of spores or substances through apertures of microscopic or macroscopic dimensions.
Organism an individual life form, usually of microscopic nature, such as a fungus.
Ostiole an opening or a pore by which spores are dispersed from a sexual or asexual fruiting body.
Ovary female reproductive structure containing an ovule or egg.
Ovate - ovoid egg-shaped, with the narrower end at the top.
Overwinter survival of a fungus from one season to the other.
Panicle an indeterminate inflorescence, the main axis of which is branched, with pedicillate flowers on the secondary branches.
Papillate having one or more, small, nipple-shaped elevations.
Paraboloid bowel-shaped.
Paraphysis [(pl=paraphyses) of fructifications] sterile filaments having free tips, usually clavate or filiform.
Parasite an organism living upon another living organism and drawing its food from it, with or without fatal effect on the host.
Pathogen an agent that causes disease.
Pathogenicity the ability of a pathogen to cause disease.
Pedicel a slender stalk bearing a spore.
Pedicellate being produced on a pedicel.
Perfect state sexual part of the life cycle of the fungus during which spores are formed as a result of some sort of sexual process; sexual stage in which a fungus is capable of sexual reproduction.
Perithecium a flask-shaped sexual fruiting body of a fungus with a pore at the top.

Phialide an end cell of a conidiophore which is usually bottle-shaped with one or more open ends on which conidia are produced.
Phytosanitary refers to the health of plants or plant commodities to be exported.
Pionnote spore mass collected at the tip of a conidiophore.
Plant quarantine official precautions used to prevent the introduction of plant pathogens and pests from one area to another usually noninfested area.
Pluriseptate see multiseptate.
Primary inoculum the first inoculum of a pathogen available for infection of the host.
Preventive a precautionary measure to prevent the occurrance of infection, for example, application of fungicides.
Propagule any part of an organism capable of initiating independent growth, such as spore, sclerotium, mycelial fragments, etc.
Protuberant having a short projection.
Pseudoparanchyma a thick tissue formed by hyphae becoming twisted and fixed together like a mass; having lost their individuality.
Pseudoseptate the appearance of having septa which do not reach across the conidium from wall to wall.
Pseudothecium (pl= pseudothecia) perithecium-like fruiting body containing asci and ascospores.
Pycnidium the sporocarp of the Sphaeropsidales, frequently globose or flask-shaped.
Pyriform pear-shaped.
Radiate spreading from a center.
Reniform kidney-shaped.
Saccate like a sac or bag.
Scar that part of the septum involved in secession which forms the base of the conidium.
Sclerotium (pl=sclerotia) a hard, dark and pigmented resting body of a fungus, comprising afungal tissue.
Seed the grain or mature ovule of a plant, used for sowing.
Seed certification certification based on official inspection and testing of seed with regard to its quality and origin.
Seed coat the outer protective cover of the seed.
Seed mycoflora refers to the association of fungi with seeds, which may or may not have the potential of causing diseases of the seed or plant.
Seed rot seed decay caused by pathogenic fungus.
Seed transmission an act of perpetuation of a pathogen from one generation to another through the seed.
Seedborne an organism or an infectious agent which is carried in, on or with the seed, but not necessarily transmitted.
Seedborne pathogen an infectious agent associated with seed and having the potential of causing a disease.
Septum (pl=septa) a cross-wall in a hypha or conidium.
Sessile without a stalk or stem.
Seta (pl=setae) stiff hair, usually dark and thick-walled.
Sign any indication of disease other than reaction of the host plant; i.e., spores, mycelium, exudate, or fruiting bodies of the pathogen.
Sorus (pl=sori) a fruiting structure in rusts and smuts; mass of spores.
Smut a plant disease caused by the fungi belonging to Ustilaginales; a smut fungus.
Solitary arising singly at one point.
Species a taxonomic division of a genus.

Spindle-shaped fusoid; narrowing towards the ends.
Spore a general term for a reproductive structure in fungi.
Spore ball an aggregate of many spores, or a group of spores clustered together usually formed in smut fungi.
Sporodochium a cushion-shaped fruiting body consisting of conidiophores and conidia.
Stem rot decay of the stem caused by a pathogenic fungus.
Sterigmata a short, pointed, extension of a cell which bears a spore. Sterigmata in one row are uniseriate, and in two rows are biseriate.
Stereobinocular microscope a kind of microscope with lenses arranged so that the object in view is seen in three dimensions.
Sterile an organism that does not produce spores.
Stipitate having a stalk.
Striate marked with lines or groves or ridges.
Stroma (pl=stomata) a matrix of vegetative hyphae, with or without host tissue, sometimes sclerotium-like in form, in or on which spores are produced.
Subclavate club-shaped with little narrowing in the direction of the base.
Subcylindric nearly cylindrical.
Subepidermal occurring just beneath the epidermis.
Subglobose almost globose or spherical.
Subhyaline somewhat or imperfectly clear or nearly colorless.
Subovoid nearly egg-shaped.
Subulate slender and tapering to a point awl shaped.
Superficial occurring on the surface of the substrate.
Supernatant clear liquid floating on the surface.
Symptom a visible expression by a host of a diseased condition.
Teliospore thick-walled resting spore produced by rust fungi.
Terminal conidium borne at the apex of a conidiophore.
Tissue an aggregate of cells usually similar in structure.
Tropical the regions which lie between the Tropics of Cancer and Capricorn.
Tympaniform drum-shaped
Unicellular one-celled.
Unilocular single, simple, undivided cavity.
Uniseriate arrangement of ascospores in an ascus in one series.
Unitunicate an ascus in which both the inner and outer walls are more or less rigid.
Velvety cushion-like.
Ventral front or lower surface.
Verrucose warty.
Verticillate arrangement of hyphae or conidiophores in whorls.
Vesicle a bladder-like or swollen apex of a conidiophore.
Vesitgial an organ having little or no utility but performing a useful function.
Violently forcefully.
Visual examination a method of examining seeds with or without the help of a magnifying lens.
Warty rough surfaced.
Whorled referring to the arrangement of conidia on the conidiophore in which more than one conidia are produced at a given locus.
Wilt a vascular plant disease caused by a fungus affecting the normal uptake and distribution of water.
Zonate marked with zones; having concentric rings.

(Source: Ainsworth et al. 1971; Dalal-Clayton 1981; Holliday 1989; and Snell and Dick 1971)

About ICRISAT

The semi-arid tropics (SAT) encompasses parts of 48 developing countries including most of India, parts of southeast Asia, a swathe across sub-Saharan Africa, much of southern and eastern Africa, and parts of Latin America. Many of these countries are among the poorest in the world. Approximately one sixth of the world's population lives in the SAT, which is typified by unpredictable weather, limited and erratic rainfall, and nutrient-poor soils.

ICRISAT's mandate crops are sorghum, pearl millet, finger millet, chickpea, pigeonpea, and groundnut; these six crops are vital to life for the ever-increasing populations of the semi-arid tropics. ICRISAT's mission is to conduct research which can lead to enhanced sustainable production of these crops and to improved management of the limited natural resources of the SAT. ICRISAT communicates information on technologies as they are developed through workshops, networks, training, library services, and publishing.

ICRISAT was established in 1972. It is one of 18 nonprofit, research and training centers funded through the Consultative Group on International Agricultural Research (CGIAR). The CGIAR is an informal association of approximately 50 public and private sector donors; it is co-sponsored by the Food and Agriculture Organization of the United Nations (FAO), the World Bank, and the United Nations Development Programme (UNDP).

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