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Coprophilous fungi from Brazil: updated identification keys to all recorded species

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Abstract

Taxonomic records of coprophilous fungi from Brazil are revisited. In total, 271 valid species names, including representatives of Ascomycota (187), Basidiomycota (32), Kickxellomycota (2), Mucoromycota (45) and Zoopagomycota (5), are reported from herbivore dung. Identification keys for coprophilous fungi from Brazil are provided, including both recent surveys (2011–2019) and historical literature.

Keywords: Agaricales, dung fungi, Mucorales, taxonomy

Introduction

Fungi able to germinate, live and feed on herbivore dung form a restricted group of microorganisms, commonly referred to as coprophilous fungi (Bell 2005, Kirschner *et al.* 2015). This ecological group can include highly specialized species that can survive the harse environment of an animal's gastrointestinal tract, symbionts in an animal's digestive tract or even generalists, non-specialized species, able to efficiently exploit these substrates (Richardson 2001b). These fungi represent an important component of ecosystems, responsible for recycling the nutrients in animal dung, and provide an important resource for experimental ecology (Krug *et al.* 2004), paleontology (Basumatary & McDonald 2017) and palynology (Florenzano 2019) studies.

Despite the great diversity of biomes and vegetation types, the study of Brazilian coprophilous fungi has received little attention. Early studies, conducted mainly by European naturalists in the 19th century, produced only a few records of these organisms (Fidalgo 1962). Augusto Chaves Batista, a reknowned Brazilian mycologist, studied the composition of yeasts and filamentous fungi of domestic, wild and captive animals, with interest in fungi with medical importance, especially yeasts (Batista *et al.* 1961). During a visit to Brazil in 1998, Richardson (2001a) identified 32 species from material collected in Mato Grosso do Sul. Alves *et al.* (2002) identified eleven taxa of *Mucor* (Mucorales, Mucoromycota) on herbivore dung in Recife (Pernambuco). In the same city, Santiago *et al.* (2011) identified 39 taxa of zygosporic fungi on dung of captive herbivores, providing an identification key. Viriato (2008) identified 10 species of *Pilobolus* in a Zoological Park in São Paulo, and Melo *et al.* (2011) recorded 22 species of coprophilous ascomycetes on captive wild herbivore dung in a Zoological Park in Recife, providing illustrations, descriptions and an identification key. Recently, more focused works on the most common genera were conducted, and new species and records have been presented (Melo *et al.* 2014, 2015a, 2015b, 2017a, 2017b, 2019). Calaça *et al.* (2014) provided a preliminary checklist of coprophilous fungi and Myxomycetes from Brazil, as well as other contributions to the knowledge of the coprophilous mycobiota of the Brazilian Cerrado (Calaça *et al.* 2013, Calaça & Xavier-Santos 2014, 2016, Calaça *et al*. 2015). Other records include scattered citations in books, monographies and revisions, usually not focused on dung fungi or on the Brazilian coprophilous mycobiota.

This study presents the available data on fungi sporulating on (or isolated from) herbivore dung recorded in Brazil based on recent studies and a critical review of the literature as well as detailed and updated identification keys.

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Material and Methods

An intensive literature search for records of coprophilous fungi from Brazil was conducted in order to compile all of the available information. The Table 1 summarizes the main literature used for identification and species confirmation purposes, providing the most relevant literature used for the identification of coprophilous fungi among its most common genera in Brazil. The taxonomic classification of species and groups followed the online databases *Index Fungorum* (IFS) (www.indexfungorum.org), the *Mycology Collections data Portal*, *MyCoPortal* (www.mycoportal.org), *Species Fungorum* (www.speciesfungorum.org) and *MycoBank* (www.mycobank.org), except when more recent reviews were available. For genera with minimal variability between species that are usually morphologically cryptic such as *Aspergillus* and *Fusarium*, a "cf." mark was added to indicate that the identification refers to the species complex/clade, but not necessarily to the most recent concept of the given species. Records without identification at the species level were not considered. For the purpose of this study, the yeasts were not considered, because: (1) they do not present morphological characters that, not allied to molecular and/or biochemical tests, can lead to their identification, and (2) because they present considerable variation according to the health of animal or the general composition of mycobiota, being more used as health indicators in veterinary medicine than ecological indicators in biodiversity studies.

Some characters presented were obtained from isolation in one of the following culture media: Malt Extract Agar (MEA) for Mucoromycota, *Cladosporium* and *Trichoderma*; Czapek Yeast Agar (CYA) for *Aspergillus*, *Penicillium* and *Paecilomyces* and Potato Dextrose Agar (PDA) for *Fusarium* and *Monascus*. Cultures were incubated at room temperature (28 ± 2 °C) for seven days under natural alternating light and dark periods (12/12 hours), unless stated otherwise. Synnematous conidial fungi were mounted in lactophenol with cotton blue and measured after removal from the dung surface.

TABLE 1. Consulted literature for the identification of coprophilous fungi species from Brazil compiled in this work.

Genera	Worldwide records
Absidia	Hoffmann *et al.* (2007), Souza *et al.* (2017)
Acremonium	Gams (1971), Gräfenhan *et al.* (2011), Summerbell *et al.* (2011)
Arnium	Cain & Mirza (1972), Lundqvist (1972)
Arthrobotrys	Cooke & Godfrey (1964), Haard (1968)
Ascobolus	Brummelen (1967), Melo *et al.* (2014)
Ascodesmis	Obrist (1961), Brummelen (1981)
Ascotricha	Ames (1961)
Aspergillus	Klich (2002), Samson *et al.* (2014), Visagie *et al.* (2014a)
Backusella	Benny & Benjamin (1976), Ellis & Hesseltine (1969)
Bolbitius	Amandeep *et al.* (2013)
Cephaliophora	Ruszkiewicz-Michalska *et al.* (2017)
Cephalotrichum	Seifert *et al.* (1983), Seifert *et al.* (2011)
Cercophora	Lundqvist (1972), Melo *et al.* (2019)
Chaetomium	Ames (1961), Arx *et al.* (1986), Melo *et al.* (2019)
Chlorophyllum	Singer (1986), Ge *et al.* (2018)
Circinella	Hesseltine & Fennell (1955)
Cladosporium	Bensch *et al.* (2010), Bensch *et al.* (2012), Melo *et al.* (2017a)
Coniochaeta	Malloch & Cain (1970), Asgari *et al.* (2007)
Conocybe	Singer (1986), Doveri (2004)
Coprinellus	Uljé & Bas (1992), Doveri (2004)
Coprinopsis	Uljé & Noordeloos (1999), Doveri (2004)
Coprinus	Uljé & Bas (1992), Doveri (2004)
Coprotus	Kimbrough *et al.* (1972), Doveri (2004), Melo *et al.* (2015b)
Cunninghamella	Zheng & Chen (2001)

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TABLE 1. (Continued)

Genera	Worldwide records
Cyathus	Brodie (1975), Cruz *et al.* (2014)
Dactylella	Cooke & Godfrey (1964)
Dactylellina	Cooke & Godfrey (1964)
Delitschia	Luck-Allen & Cain (1975), Calaça *et al.* (2015)
Dimargaris	Benjamin (1959)
Dispira	Benjamin (1959)
Drechmeria	Cooke & Godfrey (1964), Samuell *et al.* (2000)
Drechslerella	Cooke & Godfrey (1964)
Faurelina	Locquin-Linard (1975), Arx *et al.* (1988)
Fusarium	Booth (1971), Smith (2007)
Gilbertella	Benny (1991) Santiago & Cavalcanti (2007)
Graphium	Jacobs *et al.* (2003), Lackner & de Hoog (2011)
Harposporium	Cooke & Godfrey (1964), Chaverri *et al.* (2005), Wang *et al.* (2007)
Hypocopra	Calaça & Xavier-Santos (2016)
Iodophanus	Doveri (2004), Cinto *et al.* (2007)
Kernia	Malloch & Cain (1971b)
Lasiobolus	Bezerra & Kimbrough (1974)
Lecythophora	Perdomo *et al.* (2011)
Leuconeurospora	Malloch & Cain (1970), Arx *et al.* (1988)
Lichtheimia	Hoffmann *et al.* (2007)
Lophotrichus	Ames (1961), Seth (1971)
Melanospora	Cannon & Hawksworth (1982), Vakili (1989)
Microascus	Sandoval-Denis *et al.* (2015)
Monascus	Hawksworth & Pitt (1983), Stchigel *et al.* (2004)
Mucor	Schipper (1973, 1975, 1978)
Myceliophthora	Arx (1975), Arx *et al.* (1988)
Mycoarachis	Malloch & Cain (1970), Rossman *et al.* (1999)
Nematoctonus	Cooke & Godfrey (1964), Samuell *et al.* (2000)
Paecilomyces	Samsom 1974, Houbraken *et al.* 2010, Melo *et al.* 2017a
Panaeolus	Ola'h (1969)
Parascedosporium	Lackner & de Hoog (2011), Melo *et al.* 2017a
Parasola	Uljé & Bas (1991), Doveri (2004)
Penicillium	Pitt 2000, Visagie *et al.* 2014b, Melo *et al.* 2017a
Phaeostilbella	Tulloch 1972, Melo *et al.* (2017a)
Phomatospora	Senanayake *et al.* (2016)
Pilaira	Nand & Mehrotra (1968), Mil'ko (1970)
Pilobolus	Hu *et al.* (1989), Nand & Mehrotra (1968)
Piptocephalis	Gräfenhan (1998)
Podosordaria	Krug & Cain (1974)
Podospora	Mirza & Cain (1969), Lundqvist (1972), Melo *et al.* (2015a)
Poronia	Jong & Rogers (1969)
Protostropharia	Cortez & Coelho (2004)
Pseudallescheria	Arx *et al.* (1988)
Psilocybe	Gúzman (1978, 1983, 1995)
Pyxidiophora	Calaça & Xavier-Santos (2016)

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TABLE 1. (Continued)

Genera	Worldwide records
Rhizopus	Zheng *et al.* (2007)
Saccobolus	Brummelen (1967), Melo *et al.* (2014)
Sarocladium	Gams (1971), Gräfenhan *et al.* (2011), Summerbell *et al*. (2011)
Schizothecium	Lundqvist (1972), Bell & Mahoney (1995)
Scutellinia	Schumacher (1990)
Selinia	Khan & Krug (1989b), Rossman *et al.* (1999)
Sordaria	Lundqvist (1972)
Sphaeronaemella	Upadhyay (1981) Weber & Webster (1997, 1998)
Sporormiella	Ahmed & Cain (1972), Bell (2005), Melo *et al.* (2017b)
Stropharia	Cortez & Coelho (2004), Cortez & Silveira (2007), Silva *et al.* (2008)
Syncephalastrum	Benjamin (1959)
Syncephalis	Ho & Benny (2007), Melo *et al.* (2016), Lazarus *et al.* (2017)
Talaromyces	Yilmaz *et al.* (2014), Melo *et al.* (2017a)
Thamnostylum	Benny & Benjamin (1975)
Thelebolus	Richardson & Watling (1997), Hoog *et al*. (2005), Doveri (2007)
Thielavia	Malloch & Cain (1973)
Trichoderma	Samuels *et al.* (2012), Chaverri *et al.* (2015)
Trichurus	Ellis (1971), Seifert *et al.* (1983)
Tripterosporella	Mirza (1968), Subramanian & Lodha (1968), Doveri (2004)
Xenoacremonium	Lombard *et al.* (2015)
Zopfiella	Lundqvist (1972), Guaro *et al.* (1991)
Zygopleurage	Lundqvist (1969)

Results

A total of 303 fungal species are reported from dung substrates in Brazil, which excluding yeasts and doubtful records, comprises 271 species distributed among 95 genera, 44 families, 19 orders and five phyla.

Among the zygosporic fungi (Subkingdoms Basidiobolomyceta, Mucoromyceta and Zoopagomyceta), three phyla have records on herbivore dung in Brazil. Mucoromycota, with 12 genera, was represented by seven families of Mucorales, among which *Pilobolus* is the most frequent.

Ascomycota has the highest number of species recorded (187), and it is in this group that coprophilous species are most well dispersed among different evolutionary lineages. Considering both anamorphic and teleomorphic states, ascomycetes from 66 genera in 29 families and 15 orders were recorded on dung in Brazil. Most genera are included in Sordariales (12), followed by Hypocreales (11) and Microascales (8). Twenty-five genera of conidial ascomycetes are recoded in Brazil, some studied in works on nematophagous fungi (Saumell & Padilha 2000, Saumell *et al.* 1999, 2000).

Thirteen genera and 32 species of Basidiomycota have been recorded on dung in Brazil, in six families of Agaricales, including both gymnocarpic and angiocarpic fungi.

Four identification keys, to species of (1) zygosporic fungi, (2) conidial fungi, (3) Basidiomycota and (4) Ascomycota, whose occurrence on dung from Brazil was reported and confirmed in this work, are presented.

Keys to coprophilous fungi from Brazil

1. Reproductive structures with hyaline to dark coloured sporangia and/or sporangiola, merosporangia, on unbranched to strongly branched hyphae, usually aseptate, with one or more one-celled hyaline to lightly coloured spores inside .... **Key 1: Zygosporic fungi**
- Reproductive structures simple or complex, with plectenchymatous fruit bodies free or inside stromata and, when simple, never with spores formed inside closed sporangia, one- to many-celled 2

2. Reproductive structures simple, composed of conidiophores or in conidiomata. Sexual spores absent .... **Key 2: Conidial fungi**
- Reproductive structures complex, with plectenchymatous fruit bodies. Sexual spores present 3
3. Sexual spores one-celled, formed on basidia. Fruit bodies usually macroscopic and conspicuous. Conidial morph absent or rare .... **Key 3: Basidiomycota**
- Sexual spores one- to many-celled, formed inside asci. Fruit bodies varying greatly in size, but usually small and occasionally inconspicuous, free or in stromata. Conidial morph common, consisting of free conidiophores or synnematous conidiomata .... **Key 4: Ascomycota**

Key 1: Zygosporic fungi (Mucoromycota, Kickxellomycota and Zoopagomycota)

1. Spores in sporangiola, with or without the formation of sporangia 2
- Sporangiospores exclusively in sporangia or in multispored merosporangia 6
2. Sporangiospores formed in apical, globose sporangia and in single and/or multispored sporangiola, borne from branches of the sporangiophore main axis .... *Backusella lamprospora*
- Sporangiospores formed otherwise 3
3. Sporangiola multispored, formed in fascicles borne on one to many points along the sporangiophore, which ends in a terminal sporangium .... *Thamnostylum pyriforme*
- Sporangiola one-spored, formed in fertile apical vesicles of the sporangiophoral main axis and its branches .... (*Cunninghamella*) 4
4. Colonies light yellow, occasionally grayish-yellow. Sporangiola large, up to 32 × 16 µm or 17.5 µm diam .... *Cunninghamella blakesleeana*
- Colonies light grey. Sporangiola smaller, up to 11 µm diam 5
5. Sporangiophores usually branched, with globose, subglobose to ovoid vesicles. Sporangiospores hyaline .... *Cunninghamella elegans*
- Sporangiophores simple to poorly branched, with ovoid, subglobose to clavate vesicles. Sporangiospores dark brown .... *Cunninghamella phaeospora*
6. Merosporangia present 7
- Merosporangia absent 14
7. Merosporangia with up to two spores 8
- Merosporangia with three or more spores 11
8. Merosporangia in fertile heads at the ends of successive dichotomously branched sporangiophores .... (*Piptocephalis*) 9
- Merosporangia in successive buddings at the ends of verticillate branches of sporangiophores 10
9. Mature fertile heads dry .... *Piptocephalis lepidula*
- Mature fertile heads with wet spore drop .... *Piptocephalis lemonnieriana*
10. Sporophores with numerous sterile branches, septa without lenticular cavities, sporangiospores remaining dry after release .... *Dispira cornuta*
- Sporophores without sterile branches, septa with lenticular cavities, spores immersed in liquid at maturity .... *Dimargaris bacillispora*
11. Sporangiophores branched, usually scattered, growing on the substrate .... *Syncephalastrum racemosum*
- Sporangiophores simple and/or gregarious, growing on a fungal host .... (*Syncephalis*) 12
12. Sporangiophores bent, with conspicuous swellings .... *Syncephalis cornu*
- Sporangiophores straight, with similar diameter along their length 13
13. Merosporangia obliquely arranged at the sporangiophoral vesicle .... *Syncephalis obliqua*
- Merosporangia apically arranged at the sporangiophoral vesicle .... *Syncephalis clavata*
14. Sporangia thick-walled, black, persistent, detaching from sporangiophores prior to their rupture 15
- Sporangia thin-walled, hyaline to faintly colored, collapsing, not detaching from sporangiophores 26
15. Sporangiophores borne from trophocysts, with conspicuous subsporangial vesicles, actively releasing sporangia .... (*Pilobolus*) 16
- Sporangiophores lacking trophocysts and subsporangial vesicles, passively releasing sporangia ... *Pilaira anomala*
16. Sporangiospores mostly ellipsoid 17
- Sporangiospores mostly globose to subglobose or ovoid to subglobose 21
17. Sporangia distinctly umbonate .... *Pilobolus umbonatus*
- Sporangia otherwise 18
18. Sporangiophores up to 0.95 mm high, sporangiospores hyaline, short-ellipsoid, 8–12 × 6–8 µm .... *Pilobolus pullus*
- Sporangiophores more than 1 mm high, sporangiospores hyaline to yellowish, ellipsoid, variable in size 19
19. Columellae plane-convex, dark gray, sporangiospores ellipsoid, 3–7.5 × 2–6 µm .... *Pilobolus roridus*
- Columellae nipple-shaped to conical, sporangiospores ellipsoid to ovoid, bigger 20
20. Columellae nipple-shaped, sporangiospores hyaline, 7.5–10 × 5–6.5 µm .... *Pilobolus crystallinus*
- Columellae campanulate to conical, sporangiospores yellowish to faintly orange, 12.5–15(–25) × 7.5–10 µm .... *Pilobolus kleinii*
21. Sporangiospores thick-walled, wall 1–1.5 µm thick .... *Pilobolus oedipus*
- Sporangiospores thin-walled, wall less than 1 µm thick 22
22. Trophocysts very long, growing almost parallel to the substrate, up to 1800 µm in length .... *Pilobolus longipes*
- Trophocysts shorter, otherwise buried in the substratum, up to 610 µm in length 23
23. Sporangiophores up to 1 mm in length, sporangiospores 3.5–4 µm diam .... *Pilobolus nanus*

- Sporangiophores and sporangiospores bigger 24
24. Sporangiospores up to 18 µm in diam ....*Pilobolus lentiger*
- Sporangiospores smaller....25
25. Columellae cylindrical, occasionally slightly constricted, 140–230 × 130–300 µm, sporangiospores globose to subglobose, 5.5–7 µm diam.....*Pilobolus minutus*
- Columellae conical, 160–180 × 185–205 µm, sporangiospores ovoid to subglobose 7–9.5 × 6–8 µm....*Pilobolus hyalosporus*
- Columella conical, not constricted, 120–175 × 150–195 µm, sporangiospores variable in size, 7.5–17 µm diam ....*Pilobolus lentiger*
26. Sporangiophores with apophysate sporangia 27
- Sporangiophores without apophysate sporangia 34
27. Sporangiophores arising from aerial mycelia or from stolons commonly with opposite rhizoids, light brown to dark brown, usually simple .... (*Rhizopus*) 28
- Sporangiophores arising from aerial mycelia or from stolons never with opposite rhizoids, hyaline to weakly pigmented, usually branched 29
28. Rhizoids profuse and strongly branched, sporangiophores up to 2.7 (–4) mm long....*Rhizopus stolonifer*
- Rhizoids weakly branched, sporangiophores up to 1.5 (–2.5) mm lomg ....*Rhizopus arrhizus*
29. Mesophilic species, not growing above 40 °C, suspensors of zygosporangia with appendages....(*Absidia*) 30
- Thermotolerant species, able to grow above 40 °C, suspensors of zygosporangia without appendages ....(*Lichtheimia*) 31
30. Sporangiospores cylindrical ....*Absidia cylindrospora*
- Sporangiospores mostly oval, some subglobose ....*Absidia repens*
31. Sporangiophores with short, hemispheric columella, never spatulate, without projections ....*Lichtheimia brasiliensis*
- Sporangiophores with globose, oval, spatulate columella, with one to several projections 32
32. Sporangia in shades of brown when mature, sporangiospores rough, pigmented....*Lichtheimia hyalospora*
- Sporangia pale yellow to pale brown, sporangiospores smooth, hyaline....33
33. Sporangiospores mostly globose to subglobose, occasionally irregular in shape ....*Lichtheimia corymbifera*
- Sporangiospores mostly ellipsoid to cylindrical, occasionally subglobose....*Lichtheimia ramosa*
34. Sporangiophores sympodially branched, bearing circinate branches, single or in umbels (*Circinella*) 35
- Sporangiophores umbranched, or branched, if so, weakly sympodially branched, not circinate, not in umbels....36
35. Sporangiophores with 1–2 sporangia, with a conspicuous sterile spine close to each sporangium....*Circinella muscae*
- Sporangiophores with 8–10 sporangia in umbels, lacking sterile spines ....*Circinella umbellata*
36. Sporangia persistent, with a longitudinal dehiscence, splitting almost at its half, sporangiospores with hyaline appendages....*Gilbertella persicaria*
- Sporangia persistent or collapsing, never splitting as above, sporangiospores lacking hyaline appendages.... (*Mucor*) 37
37. Zygospores formed from monosporic culture ....*Mucor genevensis*
- Zygospores not formed from monosporic culture....38
38. Colonies on MEA with delicate, fragile appearance, sporangiospores with one or more granules 39
- Colonies on MEA with vigorous, wooly appearance, sporangiospores without granules 40
39. Sporangiophores constricted near the columella, columella globose to ellipsoid.... *Mucor subtilissimus*
- Sporangiophores not constricted, columella elongated-conical ....*Mucor guilliermondii*
40. Mature sporangia with persistent wall....41
- Mature sporangia collapsing early....43
41. Sporangiophores with short and long branches, forming successive sympodial branches ....*Mucor ramosissimus*
- Sporangiophores unbranched or branched once....42
42. Columella globose to obovoid, sporangiospores varying considerably in shape (globose, ovoid, cylindrical or fusoid) and size ....*Mucor variosporus*
- Collumellae conical to ellipsoid, sporangiospores regular in shape and size, broadly ellipsoid to ellipsoid, rarely cylindrical ....*Mucor mousanensis*
43. Sporangia more than 150 µm in diameter 44
- Sporangia less than 150 µm in diameter....45
44. Sporangiospores broadly ellipsoid to ellipsoid, up to 10 µm long ....*Mucor piriformis*
- Sporangiospores ellipsoid to cylindrical, more than 10 µm long ....*Mucor mucedo*
45. Sporangiophores unbranched or rarely sympodially branched 46
- Sporangiophores repeatedly sympodially branched....47
46. Colonies on MEA with reverse bright yellow, strongly pigmented, columella globose, sporangiospores long-ellipsoid to fusoid.... *Mucor luteus*
- Colonies on MEA with reverse cream to white, not pigmented, columella subglobose to ellipsoid, sporangiospores ellipsoid .... *Mucor hiemalis*
47. Sporangiophores sympodially and monopodially branched, chlamydospores abundant in sporangiophores and/or in columella, sporangiospores globose to ellipsoid.... *Mucor racemosus* f. *racemosus*
- Sporangiophores successive sympodial branched, chlamydospores, if present, never produced in sporangiophores and/or in columella, sporangiospores ellipsoid, subglobose or irregular 48
48. Sporangiospores smooth-walled....49
- Sporangiospores verrucose or with minute protusions....51
49. Sporangiospores irregular in size and shape, ellipsoid, a few globose, some aberrant in size and shape....50

- Sporangiospores regular in size and shape, subglobose .... *M. circinelloides* f. *janssenii*
50. Sporangiospores ellipsoid, a few globose, up to 6 × 4.5 µm .... *M. circinelloides* f. *griseo-cyanus*
- Sporangiospores variable in shape, ellipsoid, frequently aberrant, up to 18 × 12 µm .... *M. circinelloides* f. *lusitanicus*
51. Sporangiophores sympodially or monopodially branched, columella frequently with projections .... *Mucor plumbeus*
- Sporangiophores sympodially branched, columella infrequently with small spines .... *Mucor fuscus*

KEY 2. CONIDIAL FUNGI

1. Reproductive structures on nematodes, either as a host or preyed upon, somatic mycelium usually with adhesive projections or other specialized nematode-trapping structures 2
- Reproductive structures not on nematodes, formed either from aerial mycelia or on dung material, somatic mycelium lacking nematode-trapping structures 23
2. Parasites that adhere to the nematode surface 3
- Predators that capture nematodes using traps 9
3. Conidiophores verticillate, forming tufts of phialides, conidia somewhat triangular, 3.5–4.5 × 1.5–2 µm .... *Haptocillium balanoides*
- Conidiophores with branching pattern, phialides and conidia not as above 4
4. Conidia phragmosporic, conidiogenous cells sessile .... (*Cephaliophora*) 5
- Conidia amerosporic, conidiogenous cells on conidiophores 6
5. Conidia 4–6 septate, with synchronous growth, pyriform to obtriangular, not constricted at the septa 21–36 × 12–25 µm .... *Cephaliophora tropica*
- Conidia 3–7 septate, usually with asynchronous growth, pyriform to clavate, slightly constricted at the septa, 30–60 × 14–20 µm .... *Cephaliophora irregularis*
6. Conidia lacrimoid, formed in sterigmata .... *Drechmeria coniospora*
- Conidia helicoid to fusoid, formed in subglobose phialides .... (*Harposporium*) 7
7. Conidia straight to slightly curved, with a spine at one end .... *Harposporium bysmatosporum*
- Conidia strongly curved, without such spines 8
8. Conidia elongate, 6–18 × 1–2 µm, acute at both ends .... *Harposporium anguillulae*
- Conidia shorter, 4.5–9 × 1–1.5 µm, round at both ends .... *Harposporium lilliputanum*
9. Fertile hyphae with clamp connections, conidia strongly curved .... *Nematoctonus robustus*
- Fertile hyphae without clamp connections, conidia slightly or not curved 10
10. Conidia didymosporic 11
- Conidia phragmosporic 15
11. Conidia usually 2-septate, rarely 3-septate, 26–83 × 12–17 µm .... *Arthrobotrys scaphoides*
- Conidia 1-septate 12
12. Conidia obovoid, with cells distinctly different, with an apiculate basal cell and a larger apical cell, 22–32 × 12–20 µm, constricted at the septum .... *Arthrobotrys oligospora*
- Conidia different in morphology, not constricted at the septum 13
13. Conidia pyriform to oblong-pyriform, 18–27 × 8–12 µm .... *Arthrobotrys robustus*
- Conidia ellipsoid, with different measurements (larger or smaller) 14
14. Conidia frequently slightly curved, with a truncate basal cell and an acute, larger apical cell, 32–48 × 7–9.5 µm .... *Arthrobotrys dactyloides*
- Conidia rarely curved, with a rounded basal cell usually as large as the apical cell, 22–44 × 7.5–13 µm .... *Arthrobotrys musiformis*
15. Conidia elongate, cylindrical, with a globose to ellipsoid distal cell 16
- Conidia variously shaped, larger at the central cell, lacking an inflated distal cell 17
16. Conidia usually 4-septate, 35–60 × 2.2–3.2 µm .... *Dactylellina haptospora*
- Conidia 5–15 septate, 40–105 × 4–6 µm .... *Dactylella leptospora*
17. Conidia 2–4 septate, with evenly spaced septa .... *Arthrobotrys brochopaga*
- Conidia usually 4-septate, usually close to spore extremities 18
18. Conidia rhomboid to turbinate .... *Dactylellina gephyropaga*
- Conidia spindle-shaped to ellipsoid 19
19. Conidia wide, usually larger than 18 µm 20
- Conidia narrower, usually up to 18 µm in width 22
20. Conidia apically rounded, narrowing towards the truncate base .... *Arthrobotrys eudermatus*
- Conidia truncate at both ends 21
21. Conidia spindle-shaped, usually 4-septate, 41–55 × 17–26 µm .... *Drechslerella aphrobrocha*
- Conidia large-fusoid to ellipsoid, 2–4-septate, 40–75 × 18–35 µm .... *Arthrobotrys megalosporus*
22. Conidia slightly curved, 25–76 × 7–16 µm .... *Dactylella gampsospora*
- Conidia not curved, 40–60 × 11–18 µm .... *Dactylellina phymatopaga*
23. Conidiogenous cells sessile, arising from somatic hyphae 24
- Conidiogenous cells on free conidiophores or in conidiomata 27
24. Conidia 4–6 septate .... *Cephaliophora tropica*
- Conidia amerosporic 25
25. Conidia on acute phialides, mostly clustered in a sticky, mucilaginous mass .... *Sarocladium bacillisporum*
- Conidia on ampuliform to lageniform phialides, dry 26

26. Conidia on phialides or adelophialides. *Coniochaeta* teleomorph .... *Lecythophora lignicola*
- Conidia on sessile phialides. Sordariales teleomorph .... *Phialophora*-like anamorph
27. Conidiophores clustered in synnemata or in sporodochia, occasionally free 28
- Conidiophores free, not clustered in conidiomata 35
28. Conidiophores in synnemata, conidia amerosporic 29
- Conidiophores in sporodochia or free, conidia phragmosporic 33
29. Synnemata setose, with undulated to coiled sterile setae .... *Cephalotrichum cylindricum*
- Synnemata glabrous, without setae 30
30. Conidiogenous cells phialidic .... *Phaeostilbella atra*
- Conidiogenous cells annelidic 31
31. Synnemata with loosely compact spore heads, bearing dry spores .... *Cephalotrichum purpureofuscus*
- Synnemata with tightly compact spore heads, bearing wet spores 32
32. Synnemata olive green to olive brown, 175–200 µm high, without variation in pigmentation towards the apex, conidia 4–6 × 1–2.5 µm .... *Graphium penicillioides*
- Synnemata dark brown at the base, becoming less pigmented towards the apex, light brown close to the spore head and finally hyaline in some conidiophores, 350–900 µm high, conidia 6–7.5 × 2–3.5 µm .... *Parascedosporium putredinis*
33. Macroconidia delicate, usually falciform, with acute ends, occasionally forming hooks at the apex and a foot cell at the base, inconspicuous to distinct .... *Fusarium fujikuroi*
- Macroconidia robust, curved or not, with distinct extremities 34
34. Phialides long and slender, with acute apex, macroconidia rarely curved, with acute apices .... *Fusarium solani* species complex (*F.* cf. *solani*)
- Phialides short and stout, macroconidia usually curved, with rounded apices .... *Fusarium oxysporum* species complex (*F.* cf. *oxysporum*)
35. Conidiogenesis tallic-arthric, with conidia mostly bound by cytoplasmatic filaments after release .... *Neurospora sitophila*
- Conidiogenesis blastic 36
36. Conidiogenesis blastic-acropetal .... (*Cladosporium* s.l.) 37
- Conidiogenesis enteroblastic 39
37. Conidiophores nodose, with fertile intercalary and terminal swellings .... *Cladosporium cladosporioides* species complex *(C.* cf. *oxysporum)*
- Conidophores without conspicuous swellings 38
38. Conidia densely verrucose, 7.5–23 × 3–8 µm .... *Cladosporium herbarum* species complex (*C.* cf. *herbarum*)
- Conidia smooth, 3.5–7.5 × 2–5 µm .... *Cladosporium cladosporioides* species complex (*C.* cf. *cladosporioides*)
39. Conidiogenesis blastic-percurrent, with distinct annellides .... *Microascus brevicaulis*
- Conidiogenesis blastic-phialidic, with distinct phialides 40
40. Colonies with rapid growth on culture media, greater than 9 cm radius (18 cm diameter) in seven days, in shades of green, conidiophores variably branched, phialides lageniform, with inconspicuous necks .... (*Trichoderma*) 41
- Colonies with slow to moderate growth on culture media, less 9 cm radius (18 cm diameter) in seven days, variously coloured, conidiophores simple or branching at acute angles, phialides ampulliform to acerose 45
41. Conidia dark green, conspicuously tuberculate .... *Trichoderma* clade *viride* (*T.* cf. *glaucum*)
- Conidia light green to hyaline, smooth 42
42. Colonies on MEA with golden to amber soluble pigment, conidia clavate ...*Trichoderma* clade *Chlorospora* (*T.* cf. *aureoviride*)
- Colonies on MEA with soluble pigment different or absent, conidia globose to ellipsoid 43
43. Cellipsoidonidiophores with a thick main axis, forming isolated phialides and short branches along their length, conidia ellipsoid .... *Trichoderma* clade *Longibrachiatum* (*T.* cf. *pseudokoningii*)
- Conidiophores with a slender main axis, with longer branches at the base, decreasing in length towards the apex, conidia subglobose to oval 44
44. Colonies light green to pistachio green, conidiophores with densely spaced branches, phialides usually clustered, conidia globose, 2.5–3.5 × 2.5–3 µm .... *Trichoderma harzianum* species complex (*T.* cf. *harzianum*)
- Colonies dark green to gray green, conidiophores with widely spaced branches, phialides mostly single, conidia oval, 3.5–4 × 3–3.5 µm .... *Trichoderma koningii* species complex (*T.* cf. *ovalisporum*)
45. Phialides long, narrow, and tapered, conidia stuck together in a mucilaginous mass, forming a spore head above each phialide 46
- Phialdes ampuliform to acerose, conidia dry 49
46. Conidia 1- to 2-septate, falciform, 5–27 × 2.5–5 µm .... *Hyalopus keratinoplasticum*
- Conidia usually non-septate (rarely 1-septate), different in shape 47
47. Conidia reniform, 4–6 × 1.3–2 µm .... *Xenoacremonium recifei*
- Conidia distinctly shaped, cylindrical to falciform 48
48. Conidia cylindrical, 2.5–3 × 0.5–1 µm .... *Sarocladium bacillisporum*
- Conidia cylindrical to falciform, 6–25 × 2.5–6 µm .... *Hyalopus onycophilus*
49. Phialides on inflated vesicles at the conidiophore apex .... (*Aspergillus* s.l.) 50
- Phialides on non-inflated hyphae 60
50. Conidiophores with predominantly uniseriate vesicles 51
- Conidiophores with predominantly biseriate vesicles 53
51. Colonies on CYA25 dark brown to black in seven days .... *Aspergillus* clade *Japonicus-Aculeatus (A.* cf. *japonicus)*
- Colonies on CYA25 shades of green to blue or brown in seven days 52
52. Colonies on CYA25 dark green to brown in seven days, vesicles spherical to elongate, conidia distinctly rough-walled ....

.... *Aspergillus* clade *Flavus* (*A.* cf. *parasiticus*)
- Colonies on CYA25 greenish blue to blue in seven days, vesicles spathulate, conidia smooth, occasionally roughened..................
.... *Aspergillus* clade *Fumigati* (*A.* cf. *fumigatus*)
53. Colony diameter on CYA25 less than 60 mm in seven days..54
- Colony diameter on CYA25 greater than 60 mm in seven days...58
54. Sporiferous parts of colony on CYA25 shades of green... *Aspergillus versicolor*
- Sporiferous parts of colony on CYA25 not shades of green 55
55. Colonies on CYA25 and/or MEA25 whitish to cream coloured .... *Aspergillus* clade *Terrei (A.* cf. *niveus)*
- Colonies on CYA25 and/or MEA25 ochraceous to grayish...56
56. Spore heads frequently strongly columnar, bearing long chains of conidia.........................*Aspergillus clade Terrei* (*A.* cf. *terreus*)
- Spore heads frequently radiate, rarely loosely columnar, bearing short chains of conidia 57
57. Colonies on MEA25 olivaceous gray, vesicles 8–10 µm in diam...*Aspergillus variecolor*
- Colonies on MEA25 olivaceous brown, vesicles 7–16 µm in diam .... *Aspergillus ustus*
58. Colonies on CYA25 matte black to olivaceous black after seven days, conidia usually ornamented ....
....*Aspergillus* clade *Nigri* (*A.* cf. *niger*)
- Colonies on CYA25 shades of green after seven days, conidia usually smooth 59
59. Colonies on CYA25 pistachio-green to greyish green after seven days, conidial heads radiate, compact ....
....*Aspergillus* clade *flavus* (*A.* cf. *flavus*)
- Colonies on CYA25 bronze to brownish after seven days, conidial heads varying greatly in morphology, occasionally uniseriate..
.... *Aspergillus* clade *tamarii* (*A.* cf. *tamarii*)
60. Colonies brownish to cream coloured, phialides with long necks, conidia in long, sinuous chains...
.... *Paecilomyces variotii* species complex (*P.* cf. *variotii*)
- Colonies whitish to greenish, phialides with short necks, conidia in straight, short chains...61
61. Conidiophores predominantly monoverticillate .... *Penicillium glabrum* species complex (*P.* cf. *glabrum*)
- Conidiophores predominantly branched..62
62. Conidiophores predominantly terverticillate .... *Penicillium* clade *Chrysogena (P.* cf. *chrysogenum)*
- Conidiophores predominantly biverticillate 63
63. Ratio of phialide to metula length near one, reddish soluble pigments usually present ....
....*Talaromyces* clade *Talaromyces* (*T.* cf. *purpurogenus*)
- Ratio of phialide to metula length much less than one, soluble pigments, if present, not reddish..64
64. Penicilli predominantly ending in terminal verticils of metulae 65
- Penicilli with metulae of uneven length, sometimes in verticils..69
65. Colonies with rapid growth, greater than 30 mm on CYA25 after seven days 66
- Colonies with slower growth, less than 30 mm on CYA25 after seven days..68
66. Conidia (1.5–)2–3 µm wide...*Penicillium* clade *Brevicompacta-Ramosa* (*P.* cf. *raistrickii*)
- Conidia wider 67
67. Conidia ellipsoid, 3.5–7.5 × 2.5–3.5 µm .... *Penicillium* clade *Lanata-Divaricata* (*P.* cf. *oxalicum*)
- Conidia ellipsoid to rectangular, 2.8–3.3 × 2–2.5 µm ....*Geosmithia namyslowskii*
68. Colonies on CYA25 15–28 mm diameter after seven days, dark green to turquoise green, usually with strong brightly yellowish pigment...*Penicillium* clade *Citrina* (*P.* cf. *citrinum*)
- Colonies on CYA25 23–30 mm diameter after seven days, greenish gray to dull green, usually without pigments.........................
....*Penicillium* clade *Exilicaulis* (*P.* cf. *corylophilum*)
69. Conidia ellipsoid, with somewhat acute ends and roughened wall ....*Penicillium simplicissimum*
- Conidia globose, rarely subglobose, with round ends and smooth wall 70
70. Colonies on CYA25 predominantly whitish after seven days, metulae usually forming tetrads ....
....*Penicillium* clade *Canescentia* (*P.* cf. *janczewskii*)
- Colonies on CYA25 predominantly dark green to greyish green after seven days, metulae usually 3–5 per conidiophore................
.... *Penicillium* clade *Citrina* (*P.* cf. *waksmanii*)

KEY 3: BASIDIOMYCOTA

1. Basidiomata angiocarpic, nidularioid, with gleba inside distinct peridioles exposed at maturity, light brown, outer and inner surfaces smooth, 10–13 × 4–5 mm.. *Cyathus stercoreus*
- Basidiomata gymnocarpic, agaricoid, with basidia exposed in a lamellar hymenophore..2
2. Pileus deliquescing to some degree during basidioma maturation, basidia of unequal length in the same lamella 3
- Pileus not deliquescing, basidia uniform in length..17
3. Velar remnants on pileus absent in mature basidiomata, stipe and pileus with setae and/or setulae 4
- Velar remnants on pileus present in mature basidiomata, stipe and pileus lacking setae and/or setulae 9
4. Stipe and pileus glabrous, basidiospores triangular, with rounded angles ....*Parasola misera*
- Stipe and pileus with setulae and/or setae, basidiospores otherwise..5
5. Basidiospores angular...6
- Basidiospores ellipsoid, ovoid or oblong 7
6. Pileus setulose, with setulae slightly capitate, basidiospores hexagonal, 10.8–15 × 6.5–7.5 µm............... *Coprinellus marculentus*
- Pileus setose, with acute setae, basidiospores mitriform in frontal view, 7.2–8.4 × 6–7.2 µm.......................*Coprinellus angulatus*
7. Pileocystidia with rounded ends, basidiospores 9.7–13.8 × 6.7–8.8 µm ....*Coprinellus curtus*
- Pileocystidia with abrupt ends..8

8. Pilleipelis with velar sphaerocysts, basidiospores 12.5–16.5 × 7–8.5 µm .... *Coprinellus heptemerus*
- Pilleipelis without velar sphaerocysts, basidiospores 6.5–9.5 × 3–4 µm .... *Coprinellus pellucidus*
9. Velar remnants mainly composed of globose elements..10
- Velar remnants mainly composed of filamentous elements 14
10. Velar elements smooth, interspersed with small crystals 11
- Velar elements punctate, ornamented with several crystalline projections 13
11. Basidiospores limoniform in frontal view, with rounded angles, 12.2–19 × 10.8–15.6 µm .... *Coprinopsis nivea*
- Basidiospores angular in frontal view, with abrupt angles...12
12. Basidiospores 6-sided, hexagonal in frontal view, 12.5–15 × 7.5–9 µm ....*Coprinopsis cothurnata*
- Basidiospores roughly 5-sided, cordiform in frontal view, 7–8.5 × 6–7.5 µm...*Coprinus patouillardii*
13. Basidiomata with strong narcotic smell, basidiospores ovoid to oblong, 7.5–10.5 × 5–7.5 µm......................... *Coprinus foetidellus*
- Basidiomata without narcotic smell, basidiospores cylindrical, 6.5–7.5 × 3–4 µm.. *Coprinopsis stercorea*
14. Terminal cells of velar filaments undifferentiated...15
- Terminal cells of velar filaments with thickened wall...16
15. Basidiospores 7.5–9.6 × 4.8–6 µm ....*Coprinopsis pseudoradiata*
- Basidiospores 14–16 × 7.5–8.5 µm .... *Coprinopsis radiata*
16. Velar filaments with constrictions at the septa, composed of sausage-shaped strings, basidiospores 8.5–11.5 × 5.5–7.5 µm........... .... *Coprinopsis cinerea*
- Velar filaments without constrictions at the septa, composed of tubular strings, basidiospores 8–12 × 6–7 µm ....*Coprinopsis vermiculifer*
17. Basidiomata growing on incubated dung or *in situ* 18
- Basidiomata growing *in situ* on dung or in soil contaminated with dung, usually absent in incubation 22
18. Pileus convex, hemispherical to conical-campanulate when expanded, pale cream, becoming ochraceous with age, bearing free to adnate lamellae, equally copper to rust coloured when basidiospores ripening, spore print brown 19
- Pileus hemispherical when expanded, whitish, usually remaining so with age, bearing adnexed lamellae, white, markedly punctate with black dots due the non-simultaneous basidia maturation, spore print black 20
19. Basidiomata growing *in situ*, expanded pileus 36–40 mm diam...*Bolbitius demangei*
- Basidiomata growing on incubated dung, expanded pileus 9–14 mm diam ....*Conocybe siliginea*
20. Pileus umbonate, cinnamon colored to brown, with brownish context.. *Panaeolus cinctulus*
- Pileus hemispherical to campanulate, whitish to cream colored, with whitish context 21
21. Expanded pileus convex to campanulate, brownish to pinkish brown, with involute margin .... *Panaeolus papilionaceus*
- Expanded pileus conical to hemispherical, whitish to pale cream, with lacerated margin ....*Panaeolus antilarum*
22. Spore print white to whitish, lamellae whitish, with some shades of green, expanded pileus with conspicuous yellowish scales..... ....*Chlorophyllum hortense*
- Spore print dark, lamellae darkening with basidiospore maturation, never greenish, expanded pileus without yellowish scales 23
23. Chrysocystidia present...24
- Chrysocystidia absent...25
24. Partial veil fleshy, basidiospores 7–9 × 5–5.5 µm...*Stropharia coronilla*
- Partial veil glutinous, basidiospores 13–14 × 7–9 µm .... *Protostropharia alcis*
25. Partial veil absent ....*Psilocybe argentina*
- Partial veil present 26
26. Basidiomata turning blue when handled 27
- Basidiomata not turning blue when handled 29
27. Pleurocystidia absent, basidiospores ovoid ....*Psilocybe caeruleoannulata*
- Pleurocystidia present, basidiospores hexagonal 28
28. Basidiospores 13–16 × 7–10 µm .... *Psilocybe cubensis*
- Basidiospores 11–13 × 7.5–8.8 µm ....*Psilocybe subcubensis*
29. Basidiospores up to 15 µm long ....*Psilocybe coprophila*
- Basidiospores shorter, not exceeding 12 µm long...30
30. Annulus whitish, basidiospores 8.5–12 × 6–8 µm ....*Psilocybe pegleriana*
- Annulus black, basidiospores 10–11 × 6.5–7.5 µm... *Psilocybe merdaria*

KEY 4: ASCOMYCOTA

1. Ascomata apothecial, with exposed hymenium in all or in part of their development, superficial, never immersed in a stroma....... 2
- Ascomata with hymenium enclosed or exposed only by a single pore (ostiole), immersed to superficial, free or in stromata 36
2. Apothecia with reduced excipulum, inconspicuous at maturity, spores distinctly ornamented with warts or ridges, either anastomosing or forming a reticle .... (*Ascodesmis*) 3
- Apothecia with conspicuous excipulum, spores smooth or with different ornamentation..7
3. Ascospores partially or entirely covered by a perispore 4
- Ascospores not covered by a perispore 5

4. Ascospores 12.5–16 µm long, entirely covered by a perispore .... *Ascodesmis microscopica*
- Ascospores 16–21 µm long, partially covered by a perispore .... *Ascodesmis macrospora*
5. Spore ornamentation not forming ridges or anastomosing elements, consistilig of spines and short-angled ridges .... *Ascodesmis nigricans*
- Spore ornamentation inclusing anastomosing ridges 6
6. Ascospores ellipsoid, 12.5–13.5 × 7.5–9.5 µm, with numerous isolated spines and warts, occasionally anastomosing, with a conspicuous ridge, simple or branched, usually spanning entire spore length .... *Ascodesmis sphaerospora*
- Ascospores globose to subglobose, 10–12.5(–13) × 9.5–11.5(–12) µm, with ridges forming a somewhat regular reticulated pattern, with prominent spines in anastomosing ridges and spines and smaller ridges between the reticulum .... *Ascodesmis porcina*
7. Mature asci strongly prominent at the disk, giving it a punctate to furfuraceous appearance, ascospores shades of brown, vinaceous to reddish, smooth to strongly ornamented 8
- Mature asci not to weakly prominent at the disk, giving it a villose to rugose appearance, ascospores hyaline to pale coloured, smooth 22
8. Ascospores free inside the asci in all stages of its development .... (*Ascobolus*) 9
- Ascospores firmly united to form a spore cluster in all or part of its development .... (*Saccobolus*) 15
9. Apothecia eugimnohymenial, larger, up to 2 mm diam 10
- Apothecia cleistohymenial, smaller, up to 0.6 mm diam 11
10. Receptacle chestnut brown when mature. Ascospores smooth .... *Ascobolus castaneus*
- Receptacle white to pale luteous when mature. Ascospores usually with one to a few oblique fissures .... *Ascobolus scatigenus*
11. Hymenium exposed at the telohymenial phase, epispore with a pattern of subparallel crevices, occasionally anastomosing 12
- Hymenium exposed at the mesohymenial phase, epispore smooth to finely granulated, occasionally with a few spaced fissures 13
12. Receptacle with crenulated, prominent margin, epispore with a regular pattern of longitudinal fissures, ascospores 10–15 × 6.5–7.5 µm .... *Ascobolus crenulatus*
- Receptacle with finely denticulate margin, epispore with a regular pattern of thin closely spaced fissures, ascospores 22–27.5 × 12–13.5 µm .... *Ascobolus levisporus*
13. Ascospores more than 50 µm long, surrounded by a gelatinous sheath .... *Ascobolus immersus*
- Ascospores shorter, gelatinous sheath present or absent 14
14. Disk with many mature asci simultaneously exposed appearing furfuraceous, ascospores 32.5–37.5 × 15.5–17 µm .... *Ascobolus americanus*
- Disk with only a few mature asci simultaneously exposed (usually less than 20 per ascoma), not appearing furfuraceous, ascospores 27.5–32 × 12.5–15 µm .... *Ascobolus elegans*
15. Spore clusters loose, visible in a regular pattern only in immature asci, ascospores free at maturity .... *Saccobolus saccoboloides*
- Spore clusters compact, even after liberation, ascospores firmly united in all stages of ascus development 16
16. Spore clusters with four rows of two longitudinally disposed ascospores, with two longitudinal planes of symmetry 17
- Spore clusters with different arrangement pattern 20
17. Apothecia with receptacle yellow to lemon-yellow at maturity, ascospores with abruptly truncated ends, 20–23 × 8–10 µm .... *Saccobolus citrinus*
- Apothecia with receptacle golden-yellow to amber at maturity, ascospores with rounded ends 16
18. Ascospores 25–27.5 × 7.5–12.5 µm .... *Saccobolus glaber*
- Ascospores shorter, less than 20 µm 19
19. Ascospores 13–20 × 8–9.5 µm, spore clusters shortened at maturity .... *Saccobolus truncatus*
- Ascospores 11.5–12.5 × 5–7.5 µm, spore clusters retaining their length and arrangement pattern at maturity .... *Saccobolus minimus*
20. Spore clusters with ascospores forming straight angles in relation to their axes, with four pairs loosely united in a cylindrical pattern .... *Saccobolus infestans*
- Spore clusters with different arrangement pattern 21
21. Spore clusters with two rows with three spores and one row with two spores, the axes of the spores being parallel to the axis of the cluster .... *Saccobolus verrucisporus*
- Spore clusters as above, but the two terminal pairs of spores being parallel to the cluster axis and the two intermediate pairs obliquely arranged 22
22. Ascospores with strongly roughened epispore, warted or reticulate, 17.5–22.5 × 8.5–10 µm .... *Saccobolu beckii*
- Ascospores with smooth to finely granulate epispore, occasionally with an incomplete network of fissures, shorter than 20 µm 23
23. Spore clusters 29–32 × 9.5–12 µm, ascospores 11.5–12.5 × 6–6.5 µm .... *Saccobolus depauperatus*
- Spore clusters 38–48 × 16–20 µm, ascospores 14.5–19 × 7–10 µm .... *Saccobolus versicolor*
24. Mature apothecia glabrous 23
- Mature apothecia hairy 32
25. Apothecia reddish, pinkish to orange-yellow at maturity, asci operculate, ascospores ornamented, (17.5–)20–23(–25) × 9.5–12.5 µm .... *Iodophanus carneus*
- Apothecia pale yellow to hyaline, asci inoperculate, ascospores smooth 26
26. Apothecia yellow to luteous when young, becoming darker with maturation, usually larger than 150 µm diam., paraphyses usually inflated at the apex .... (*Thelebolus*) 27

- Apothecia hyaline to yellowish, usually not becoming darker with maturation, up to 140 µm diam., paraphyses inflated or not ..... (*Coprotus*) 28
27. Asci 8-spored. Ascospores 7.5–10 × 3–5 µm .... *Thelebolus microsporus*
- Asci with more than eight ascospores. Ascospores 6 × 3 µm .... *Thelebolus polysporus*
28. Asci multispored (with more than eight ascospores) 29
- Asci 8-spored 30
29. Asci 32-spored .... *Coprotus albidus*
- Asci 64-spored .... *Coprotus niveus*
30. Apothecia yellowish to orange, paraphyses sligthly inflated at the apex, with conspicuous orange cytoplasmatic contents, excipulum weakly to non-cyanophilous 31
- Apothecia translucid to whitish, paraphyses without conspicuous cytoplasmatic contents, excipulum strongly cyanophilous 33
31. Ascospores 16.5–18 × 10–11.5 µm .... *Coprotus ochraceus*
- Ascospores smaller 32
32. Ascospores 8.5–10 × 4–5.5 µm .... *Coprotus luteus*
- Ascospores 13–15 × 6.5–9 µm .... *Coprotus aurora*
33. Asci 65–100 × 13–16 µm, ascospores 8–10 × 6.5–7 µm .... *Coprotus lacteus*
- Asci not as above, ascospores larger 34
34. Ascospores 14–18 × 7.5–11.5 µm .... *Coprotus leucopocillum*
- Ascospores 12–13.5 × 5–8 µm .... *Coprotus discullus*
35. Ascospores 10.5–14 µm wide, with proheminent warts and ridges up to 1 µm high .... *Scutellinia scutellata*
- Ascospores smooth .... *Lasiobolus papillatus*
36. Ascomata non-ostiolate, asci usually prototunicate, evanescent 37
- Ascomata ostiolate, asci prototunicate, unitunicate or bitunicate, evanescent or persistent 51
37. Asci formed in chains, ascospores ellipsoid, ornamented .... *Talaromyces funiculosus*
- Characters above when present, not combined 35
38. Cleistothecia stipitate, on stalk-like hyphae, with peridium formed by thick hyphae fused in a loose prosenchyma .... *Monascus ruber*
- Cleistothecia sessile, on somatic hyphae, with peridium formed by a pseudoparenchyma 39
39. Ascospores with conspicuous germ pores or germ slits 40
- Ascospores without conspicuous germ pores or germ slits 48
40. Ascospores with a single, longitudinal to slightly oblique germ slit spanning entire spore length .... *Coniochaeta* sp.
- Ascospores with one or more germ pores 41
41. Ascospores with a single germ pore 42
- Ascospores with two germ pores 47
42. Ascospores with more than one cell, including an apical, dark-brown to black coloured head cell, and an hyaline basal pedicel 43
- Ascospores one-celled, subglobose to ellipsoid, hyaline to pale ochraceous 46
43. Ascospores sigmoid to cylindrical when immature, pedicels persistent, long, usually geniculate at the distal end, both apical and basal gelatinous cauda present .... *Tripterosporella pakistanii*
- Ascospores clavate when immature, pedicels usually collapsing, short or long, cylindrical, not geniculate, gelatinous cauda absent .... (*Zopfiella*) 44
44. Cleistothecia with cylindrical, dark-brown, thick-walled, septate hairs, becoming less pigmented towards the apex, pedicel 6–10 × 2.5–5 µm .... *Zopfiella erostrata*
- Cleistothecia glabrous or covered by hyphoid hairs, ascospores with pedicels of different size 45
45. Ascospores with long pedicels, 9.5–13.5 µm long .... *Zopfiella longicaudata*
- Ascospores with short pedicels, reduced to a small papillae, persistent or collapsing, up to 7.5 µm long .... *Zopfiella latipes*
46. Cleistothecia glabrous, peridium with elongated cells (*textura epidermoidea*) .... *Thielavia terrestris*
- Cleistothecia with two (occasionally three) tufts of long hairs on opposite ends, usually distally circinate, up to 600 µm long, peridial cells globose (*textura globulosa*) to angulated (*textura angularis*) .... *Kernia nitida*
47. Cleistothecia 80–110 µm diam., glabrous, with peridium formed by an outer layer with a reticulated pattern of minute ridges, ascospores 12.5–18 × 8.5–9.5 µm .... *Myceliophthora sepedonium*
- Cleistothecia 150–250 µm diam., sparsely adorned by hyphoid hairs, peridial cells smooth, ascospores 5–6.5 × 3–4 µm .... *Pseudallescheria boydii*
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Discussion

The diverse and complex coprophilous mycobiota from Brazil reflects the wide array of ecological niches, habitats, strategies and life histories in megadiverse tropical ecosystems, which have not received relative attention from researchers for long periods in recent years.

The more recent surveys on coprophilous fungi from Brazil, albeit preliminary, were less focused on single assemblages or in fungi with veterinary or human health importance, (e.g. yeasts and stress-tolerant, airborne saprobes) and thus, the aforementioned results point to a more homogenous distribution in records in Ascomycota in comparison with the others groups. Species of ascomycetes can fruit in different dung types across the country, with a great variety of forms (apothecial, perithecial, etc.) and life strategies (mycoparasitic, nematophagous, entomophilous, endocoprophilous, etc.). Zygosporic fungi are often regarded as pioneer, early occupants on dung, due to their high investment in mycelial growth and the production of asexual sporangia in the first two weeks of incubation (Richardson 2001b). In contrast, with a different mode of resource acquisition on dung, are the genera of the other two phyla, *Dimargaris* and *Dispira* (Kickxellomycota) and *Piptocephalis* and *Syncephalis* (Zoopagomycota), haustorial parasites of dung-inhabiting Mucorales and/or ascomycetes (Tretter *et al.* 2014, Lazarus *et al.* 2017). These groups are rarely registered in Brazil, reinforcing the importance of this study not only for the knowledge of coprophilous fungae, but also of understudied fungi that may represent interesting evolutionary steps. Most records of Basidiomycota are reported as "Coprinoid fungi", formerly under *Coprinus* Pers. With the segregation of this genus in 2001, most species were reclassified into two other genera: *Coprinellus* P. Karsten (1879) and *Coprinopsis* P. Karst. (1881) (Psathyrellaceae) (Redhead *et al.* 2001).

Although polyphasic approaches are currently used in fungal biology and ecology, identification guides/keys are useful in the study of mycobiotas, human resource training in fungal taxonomy, classroom use, complementary material for broad diversity studies, among others. We believe that the continued effort in the study of the underestimated fungal biodiversity, among which one of the most peculiar ecological group is addressed here, could raise a best scope for fungal taxonomy in conservation studies, management and use of biological resources.

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References

Alves, M.H., Trufem, S.F.B. & Milanez, A.I. (2002) Táxons de *Mucor* Fresen. (Zygomycota) em fezes de herbívoros, Recife, PE, Brasil. *Revista Brasileira de Botânica* 25: 147–160.
https://doi.org/10.1590/S0100-84042002000200004

Ahmed, S.I. & Cain, R.F. (1972) Revision of the genera *Sporormia* and *Sporormiella*. *Canadian Journal of Botany* 50: 419–477.
https://doi.org/10.1139/b72-061

Amandeep, K. (2013) Diversity of species of the genus *Bolbitius* (Bolbitiaceae, Agaricales) collected on dung from Punjab, India. *Mycosphere* 4 (6): 1053–1064.
https://doi.org/10.5943/mycosphere/4/6/3

Ames, L.M. (1961) *A monograph of the Chaetomiaceae*. The United States army research and development series office, Washington DC.

Arx, J.A. von. (1975) On *Thielavia* and some similar genera of Ascomycetes. *Studies in Mycology* 8: 1–31.

Arx, J.A. von, Figueras, M.J. & Guarro, J. (1988) *Sordariaceous ascomycetes without ascospore ejaculation*. Berlin, J. Cramer.

Arx, J.A. von, Guarro, J. & Figueras, M.J. (1986) *The Ascomycete genus Chaetomium*. J. Berlin, Cramer.

Asgari, B. (2007) *Coniochaeta ershadii*, a new species from Iran, and a key to well-documented *Coniochaeta* species. *Nova Hedwigia* 84: 175–187.
https://doi.org/10.1127/0029-5035/2007/0084-0175

Basumatary, S. & McDonald, H. (2017) Coprophilous fungi from dung of the Greater One-Horned Rhino in Kaziranga National Park, India and its implication to paleoherbivory and paleoecology. *Quaternary Research* 88: 14–22.
https://doi.org/10.1017/qua.2017.34

Batista, A.C., Vasconcelos, C.T., Fischman, O. & Staib, F. (1961) Fungos leveduriformes e filamentosos de fezes de bovinos, no Recife. *Boletim do Instituto de Micologia da Universidade do Recife* 325: 1–27.

Bell, A. (2005) *An illustrated guide to the coprophilous Ascomycetes of Australia*. Utrecht, CBS Biodiversity Series.

Bell, A. & Mahoney, D.P. (1995) Coprophilous fungi in New Zealand I. *Podospora* species with swollen agglutinated perithecial hairs. *Mycologia* 87: 375–396.
https://doi.org/10.1080/00275514.1995.12026544

Benjamin, R.K. (1959) The merosporangiferorus Mucorales. *Aliso* 4: 321–433.
https://doi.org/10.5642/aliso.19590402.05

Benny, G.L. (1991) Gilbertellaceae, a new family of the Mucorales (Zygomycetes). *Mycologia* 83: 150–157.
https://doi.org/10.1080/00275514.1991.12025991

Benny, G.L. & Benjamin, R.K. (1975) Observations on Thamnidiaceae (Mucorales). New taxa, new combination, and notes on selected species. *Aliso* 8 (3): 301–351.
https://doi.org/10.5642/aliso.19750803.10

Benny, G.L. & Benjamin, R.K. (1976) Observations on Thamnidiaceae (Mucorales). II. *Chaetocladium*, *Cokeromyces*, *Mycotypha*, and *Phascolomyces*. *Aliso* 8 (4): 391–424.
https://doi.org/10.5642/aliso.19760804.05

Bensch, K., Braun, U., Groenewald, J.Z. & Crous, P.W. (2012) The genus *Cladosporium*. *Studies in Mycology* 72 (1): 1–401.
https://doi.org/10.3114/sim0003

Bensch, K., Groenewald, J.Z., Dijksterhuis, J., Starink-Willemse, M., Andersen, B., Summerell, B.A., Shin, H.-D., Dugan, F.M., Schroers, H.-J., Braun, U. & Crous, P.W. (2010) Species and ecological diversity within the *Cladosporium cladosporioides* complex (Davidiellaceae, Capnodiales). *Studies in Mycology* 67: 1–94.
https://doi.org/10.3114/sim.2010.67.01

Bezerra, J.L. & Kimbrough, J.W. (1975) The genus *Lasiobolus* (Pezizales, Ascomycetes). *Canadian Journal of Botany* 53: 1206–1229.

Booth, C. (1971) *The genus Fusarium*. Kew, Commonwealth Mycological Institute.

Brodie, H.J. (1975) *The Bird's Nest Fungi*. Toronto, University of Toronto Press.
https://doi.org/10.3138/9781442632516

Brummelen, J. van. (1967) A world-monograph of the genera *Ascobolus* and *Saccobolus* (Ascomycetes, Pezizales). *Persoonia*, supplement Vol. I. Rijksherbarium, Leiden.

Brummelen, J. van. (1981) The genus *Ascodesmis* (Pezizales, Ascomycetes). *Persoonia* 11: 333–358.

Cain, R.F. & Mirza, J.H. (1972) Three new species of *Arnium*. *Canadian Journal of Botany* 50 (2): 333–336.
https://doi.org/10.1139/b72-045

Calaça, F.J.S., Delpont, M. & Xavier-Santos, S. (2015) *Delitschia gigaspora* var. *pescanii*: a new variety of coprophilous fungi from

Brazil. *Mycosphere* 6 (1): 122–126.
https://doi.org/10.5943/mycosphere/6/1/12

Calaça, F.J.S., Silva, N.C., Hirooka, Y. & Xavier-Santos, S. (2013) *Neocosmospora vasinfecta* (Nectriaceae): a new record as a fimicolous Ascomycota from Brazil. *Mycosphere Online* 4: 999–1004.

Calaça, F.J.S, Silva, N.C & Xavier-Santos, S. (2014) A checklist of coprophilous fungi and other fungi recorded on dung from Brazil. *Mycotaxon* 128 (1): 205–205.
https://doi.org/10.5248/128.205

Calaça, F.J.S. & Xavier-Santos, S. (2016) New records of coprophilous ascomycetes (Fungi: Ascomycota) from Brazil and Neotropical Region. *Check List* 12 (6): 2009.
https://doi.org/10.15560/12.6.2009

Cannon, P.F. & Hawsworth, D.L. (2008) A re-evaluation of *Melanospora* Corda and similar Pyrenomycetes, with a revision of the British species. *Botanical Journal of the Linnean Society* 84 (2): 115–160.
https://doi.org/10.1111/j.1095-8339.1982.tb00363.x

Chaverri, P., Branco-Rocha, F., Jaklitsch, W., Gazis, R., Degenkolb, T. & Samuels, G.J. (2015) Systematics of the *Trichoderma harzianum* species complex and the re-identification of commercial biocontrol strains. *Mycologia* 107 (3): 558–590.
https://doi.org/10.3852/14-147

Chaverri, P., Samuels, G.J. & Hodge, K.T. (2005) The genus *Podocrella* and its nematode-killing anamorph *Harposporium*. *Mycologia* 97 (2): 433–443.
https://doi.org/10.3852/mycologia.97.2.433

Cinto, I.E., Dokmetzian, D.A. & Ranalli, M.E. (2007) *Iodophanus carneus* and *I. testaceus* - Independent taxonomic identity or synonymy: a study of their morphology and isozymes. *Boletín de la Sociedad Argentina de Botánica* 42: 181–187.

Cooke, R.C. & Godfrey, B.E.S. (1964) A key to the nematode-destroying fungi. *Transactions of the British Mycological Society* 47 (1): 61–74.
https://doi.org/10.1016/S0007-1536(64)80081-4

Cortez, V.G. & Coelho, G. (2004) The Stropharioideae (Strophariaceae), Agaricales from Santa Maria, Rio Grande do Sul, Brazil. *Mycotaxon* 89 (2): 355–378.

Cortez, V.G. & Silveira, R.M.B. (2007) Species of *Hypholoma* (Fr.) P. Kumm. (Strophariaceae, Agaricales) in Rio Grande do Sul State, Brazil. *Acta Botanica Brasilica* 21 (3): 609–621.
https://doi.org/10.1590/S0102-33062007000300008

Cruz, R. (2014) Revision of the genus *Cyathus* (Basidiomycota) from the herbaria of northeast Brazil. *Mycosphere* 5 (4): 531–540.
https://doi.org/10.5943/mycosphere/5/4/5

Doveri F. (2004) *Fungi Fimicoli Italici: A guide to the recognition of basidiomycetes and ascomycetes living on faecal material*. Trento, Assoc. Micol. Bresadola.

Doveri, F. (2007) An updated key to coprophilous Pezizales and Thelebolales in Italy. *Mycologia Montenegrina* 10: 55–82.

Ellis, M.B. (1971) *Dematiaceous Hyphomycetes*. Kew, Commonwealth Mycological Institute.

Ellis, J.J. & Hesseltine, C.W. (1969) Two New Members of the Mucorales. *Mycologia* 61 (5): 863–872.
https://doi.org/10.1080/00275514.1969.12018810

Florenzano, A. (2019) The history of pastoral activities in S Italy inferred from palynology: a long-term perspective to support biodiversity awareness. *Sustainability* 11 (2): 1–21.
https://doi.org/10.3390/su11020404

Gams, W. (1971) *Cephalosporium-artige Schimmelpilze (Hyphomycetes)*. Stuttgart, G. Fischer.

Ge, Z.-W., Jacobs, A., Vellinga, E.C., Sysouphanthong, P., van der Walt, R., Lavorato, C., An, Y.-F. & Yang, Z.L. (2018) A multi-gene phylogeny of *Chlorophyllum* (Agaricaceae, Basidiomycota): new species, new combination and infrageneric classification. *MycoKeys* 32: 65–90.
https://doi.org/10.3897/mycokeys.32.23831

Gräfenhan, T. (1998) *Taxonomic revision of the genus Piptocephalis (Fungi)*. Greifswald, Ernst-Moritz-Arndt-Universität.

Gräfenhan, T., Schroers, H.-J., Nirenberg, H.I. & Seifert, K.A. (2011) An overview of the taxonomy, phylogeny, and typification of nectriaceous fungi in *Cosmospora*, *Acremonium*, *Fusarium*, *Stilbella*, and *Volutella*. *Studies in Mycology* 68: 79–113.
https://doi.org/10.3114/sim.2011.68.04

Guarro, J., Cannon, P.F., Aa, H.A. van der. (1991) A synopsis of the genus *Zopfiella* (Ascomycetes, Lasiosphaeriaceae). *Systema Ascomycetum* 10: 79–112.

Guzmán, G. (1978) The species of *Psilocybe* known from central and South America. *Mycotaxon* 7 (2): 225–235.

Guzmán, G. (1983) The genus *Psilocybe*. A systematic revision of the known species including the history, distribution and chemistry of the hallucinogenic species. *Beihefte zur Nova Hedwigia* 74: 1–439.

Guzmán, G. (1995) Supplement to the monograph of the Genus *Psilocybe*. *Bibliotheca Mycologica* 159: 91–141.
Haard, K. (1968) Taxonomic Studies on the genus *Arthrobotrys* Corda. *Mycologia* 60 (6): 1140–1159.
https://doi.org/10.1080/00275514.1968.12018681
Hawksworth, D.L. & Pitt, J.I. (1983) A new taxonomy for *Monascus* species based on cultural and microscopic characteristics. *Australian Journal of Botany* 31: 51–61.
https://doi.org/10.1071/BT9830051
Hesseltine, C.W. & Fennell, D.I. (1955) The genus *Circinella*. *Mycologia* 47: 193–212.
https://doi.org/10.1080/00275514.1955.12024444
Ho, H.-M. & Benny. G.L. (2007) Two new species of *Syncephalis* from Taiwan, with a key to the *Syncephalis* species found in Taiwan. *Botanical Studies* 48: 319–324.
Hoffmann, K., Discher, S. & Voigt, K. (2007) Revision of the genus *Absidia* (Mucorales, Zygomycetes) based on physiological, phylogenetic, and morphological characters; thermotolerant *Absidia* spp. form a coherent group, Mycocladiaceae fam. nov. *Mycological Research* 111 (10): 1169–1183.
https://doi.org/10.1016/j.mycres.2007.07.002
Hoog G.S. de, Göttlig, E., Platas, G., Genilloud, O., Leotta, G. & Brummelen, J. van (2005) Evolution, taxonomy and ecology of the genus *Thelebolus* in Antarctica. *Studies in Mycology* 51: 33–76.
Houbraken, J., Verweij, P.E., Rijs, A.J.M.M., Borman, A.M. & Samson, R.A. (2010) Identification of *Paecilomyces variotii* in Clinical Samples and Settings. *Journal of Clinical Microbiology* 48 (8): 2754–2761.
https://doi.org/10.1128/JCM.00764-10
Hu, F.M., Zheng, R.Y. & Chen, G.Q. (1989) A redelimitation of the species of *Pilobolus*. *Mycosystema* 2: 111–133.
https://doi.org/10.1115/1.3261961
Jacobs, K., Kirisits, T. & Wingfield, M.J. (2003) Taxonomic re-evaluation of three related species of *Graphium*, based on morphology, ecology and phylogeny. *Mycologia* 95 (4): 714.
https://doi.org/10.2307/3761947
Jong, S.C. & Rogers, J.D. (1969) *Poronia oedipus* in Culture. *Mycologia* 61 (5): 853.
https://doi.org/10.2307/3757630
Karsten, P.A. (1881) Hymenomycetes Fennici enumerati. *Acta Societatis pro Fauna et Flora Fennica* 2 (1): 1–40.
Karsten, P.A. (1879) Rysslands, Finlands och den Skandinaviska halföns Hattsvampar. Förra Delen: Skifsvampar. *Bidrag till Kännedom av Finlands Natur och Folk* 32: 1–571.
Khan, R.S. & Krug, J.C. (1989) A new species of *Selinia*. *Mycologia* 81 (4): 653–655.
https://doi.org/10.1080/00275514.1989.12025801
Kimbrough, J.W., Luck-Allen, E.R. & Cain, R.F. (1972) North American species of *Coprotus* (Thelebolaceae, Pezizales). *Canadian Journal of Botany* 50: 957–971.
https://doi.org/10.1139/b72-116
Kirschner, R., Hsu, T., Tuan, N., Chen, C-L. & Huang, S-L. (2015) Characterization of fungal and bacterial components in gut/fecal microbiome. *Current Drug Metabolism* 16 (4): 272–283.
https://doi.org/10.2174/1389200216666150812124625
Klich, M.A. & Pitt, J.I. (1988) *A laboratory guide to common Aspergillus species and their teleomorphs*. North Ryde, Commonwealth Scientific and Industrial Research Organization.
Krug, J.C., Benny, G.L. & Keller, H.W. (2004) Coprophilous fungi. *In*: Mueller, G.M., Bills, G.F. & Foster, M.S. (Orgs.). *Biodiversity of fungi*, *Inventory and monitoring methods*. Elsevier Academic Press, London.
https://doi.org/10.1016/B978-012509551-8/50024-6
Krug, J.C. & Cain, R.F. (1974) A preliminary treatment of the genus *Podosordaria*. *Canadian Journal of Botany* 52: 589–605.
https://doi.org/10.1139/b74-075
Lackner, M. & de Hoog, G.S. (2011) *Parascedosporium* and its relatives: phylogeny and ecological trends. *IMA Fungus* 2: 39–48.
https://doi.org/10.5598/imafungus.2011.02.01.07
Lazarus, K.L., Benny, G.L., Ho, H.-M. & Smith, M.E. (2017) Phylogenetic systematics of *Syncephalis* (Zoopagales, Zoopagomycotina), a genus of ubiquitous mycoparasites. *Mycologia* 109 (2): 333–349.
https://doi.org/10.1080/00275514.2017.1307005
Locquin-Linard, M. (1975) *Faurelina*, nouveau genre d'Ascomycètes (Chadefaudiellaceae?). *Revue de Mycologie* 39 (2): 125–129.
Lombard, L., van der Merwe, N.A., Groenewald, J.Z. & Crous, P.W. (2015) Generic concepts in Nectriaceae. *Studies in Mycology* 80: 189–245.
https://doi.org/10.1016/j.simyco.2014.12.002
Luck-Allen, E.R. & Cain, R.F. (1975) Additions to the genus *Delitschia*. *Canadian Journal of Botany* 53 (17): 1827–1887.

https://doi.org/10.1139/b75-212
Lundqvist, N. (1969) *Zygopleurage* and *Zygospermella* (Sordariaceae s. lat., Pyrenomycetes). *Botanska Notiser* 122: 353–374.
Lundqvist, N. (1972) *Nordic Sordariaceae s. lat.* Uppsala, Symbolae Botanicae Upsalienses.
Malloch, D. & Cain, R.F. (1970) Five new genera in the new family Pseudeurotiaceae. *Canadian Journal of Botany* 48: 1815–1825. https://doi.org/10.1139/b70-267
Malloch, D. & Cain, R.F. (1971a) New species and combinations of cleistothecial Ascomycetes. *Canadian Journal of Botany* 50 (1): 61–72.
https://doi.org/10.1139/b72-011
Malloch, D. & Cain, R.F. (1971b) The genus *Kernia*. *Canadian Journal of Botany* 49: 855–867.
https://doi.org/10.1139/b71-126
Malloch, D. & Cain, R.F. (1973) The genus *Thielavia. Mycologia* 65: 1055–1077.
https://doi.org/10.1080/00275514.1973.12019527
Melo, R.F.R., Bezerra, J.L. & Cavalcanti, M.A.Q. (2012) Diversity of coprophilus ascomycetes from captive wild animals in Dois Irmãos State Park, Brazil. *Nova Hedwigia* 94: 153–162.
https://doi.org/10.1127/0029-5035/2012/0094-0153
Melo, R.F.R., Maia, L.C. & Santiago, A.L.C.M.A. (2016) The discovery of *Syncephalis obliqua* (Zoopagomycotina, Zoopagales) in the Neotropics. *Mycotaxon* 130 (4): 1165–1169.
https://doi.org/10.5248/130.1165
Melo, R.F.R., Miller, A.N. & Maia, L.C. (2015a) The genus *Podospora* (Lasiosphaeriaceae, Sordariales) in Brazil. *Mycosphere* 6: 201–215.
https://doi.org/10.5943/mycosphere/6/2/10
Melo, R.F.R., Miller, A.N. & Maia, L.C. (2015b) *Coprotus* (Thelebolaceae, Thelebolales) in herbivore dung from Brazil. *Nova Hedwigia* 101 (1–2): 35–48.
https://doi.org/10.1127/nova_hedwigia/2014/0232
Melo, R.F.R., Miller, A.N. & Maia, L.C. (2017a) Conidial fungi associated with herbivore dung in Brazil. *Nova Hedwigia* 105 (3): 495–510.
https://doi.org/10.1127/nova_hedwigia/2017/0429
Melo, R.F.R., Miller, A.N. & Maia, L.C. (2017b) *Sporormiella longicolla* sp. nov. and new *Sporormiella* records on herbivore dung from Brazil. *Mycotaxon* 132: 459–470.
https://doi.org/10.5248/132.459
Melo, R.F.R., Monte, D.B.P., Gondim, N. H. B., Maia, L.C. & Miller, A.N. (2019) Coprophilous fungi from Brazil: New records for the Neotropics. *Mycotaxon* 134 (2): 335–352.
https://doi.org/10.5248/134.335
Melo, R.F.R, Santiago, A.L.C.M.A & Maia, L.C. (2014) The genera *Ascobolus* and *Saccobolus* (Ascobolaceae, Pezizales) in Brazil. *Mycosphere* 5: 790–804.
https://doi.org/10.5943/mycosphere/5/6/9
Mirza, J.H. (1968) *Cleistobombardia*, a new cleistothecial genus in the. family Sordariaceae. *Mycologia* 60: 704–707.
https://doi.org/10.1080/00275514.1968.12018621
Mirza, J.H. & Cain, R.F. (1969) Revision of the genus *Podospora*. *Canadian Journal of Botany* 47: 1999–2048.
https://doi.org/10.1139/b69-293
Mil'ko, A.A. (1970) *Pilaira caucasica* Milko and related species. *Mikologiya i Fitopatologiya* 4: 262–264.
Nand, K. & Mehrotra, B.S. (1968) Species of *Pilobolus* and *Pilaira* from India. *Sydowia* 22: 299–306.
Obrist, W. (1961) The genus *Ascodesmis*. *Canadian Journal of Botany* 39 (4): 943–953.
https://doi.org/10.1139/b61-079
O'Donnell, K.L. (1979) *Zygomycetes in culture*. Georgia, University of Georgia.
Ola'h, G.M. (1969) *Le genre Panaeolus: Essai taxinomique et physiologique*. Paris, Herbier Louis Marie.
Perdomo, H., Sutton, D.A., García D., Fothergill, A.W., Gené, J., Cano, J., Summerbell, R.C., Rinaldi, M.G. & Guarro, J. (2011) Molecular and Phenotypic Characterization of *Phialemonium* and *Lecythophora* Isolates from Clinical Samples. *Journal of Clinical Microbiology* 49 (4): 1209–1216.
Pitt, J.L. (1979) *The genus Penicillium and its teleomorphic states. Eupenicillium and Talaromyces*. New York, Academic Press.
Redhead, S.A., Vilgalys, R., Moncalvo, J.-M., Johnson, J. & Hopple, J.S. Jr. (2001) *Coprinus* Persoon and the disposition of *Coprinus* species sensu lato. *Taxon* 50 (1): 203–241.
https://doi.org/10.2307/1224525
Richardson, M.J. (2001a) Coprophilous Fungi from Brazil. *Brazilian Archives of Biology and technology* 44 (3): 283–289.

https://doi.org/10.1590/S1516-89132001000300010
Richardson, M.J. (2001b.) Diversity and occurrence of coprophilous fungi. *Mycological Research* 105 (4): 387–402. https://doi.org/10.1017/S0953756201003884
Richardson, M.J. & Watling, R. (1997) *Keys to Fungi on Dung*. 2nd edition. British Mycological Society. https://doi.org/10.1016/S0007-1528(68)80026-4
Rossman, A.Y., Samuels, G.J., Rogerson, C.T. & Lowen, R. (1999) Genera of Bionectriaceae, Hypocreaceae, and Nectriaceae (Hypocreales, Ascomycetes). *Studies in Mycology* 42: 1–248.
Ruszkiewicz-Michalska, M., Knysak, P., Skrobek, I., Gwiazda, A., Piskorski, S. & Żelazna-Wieczorek, J. (2017) *Cephaliophora tropica*: a third European record. *Mycotaxon* 132 (2): 445–451. https://doi.org/10.5248/132.445
Samson, R.A. (1974) *Paecilomyces* and some allied Hyphomycetes. *Studies in Mycology* 6: 1–119.
Samson, R.A., Visagie, C.M., Houbraken, J., Hong, S.-B., Hubka, V., Klaassen, C.H.W., Perrone, G., Seifert, K.A., Susca, A., Tanney, J.B., Varga, J., Kocsubé, S., Szigeti, G., Yaguchi, T. & Frisvad, J.C. (2014) Phylogeny, identification and nomenclature of the genus *Aspergillus*. *Studies in Mycology* 78: 141–173. https://doi.org/10.1016/j.simyco.2014.07.004
Samuels, G.J., Ismaiel, A., Mulaw, T.B., Szakacs, G., Druzhinina, I.S., Kubicek, C.P. & Jaklitsch, W.M. (2012) The *Longibrachiatum* Clade of *Trichoderma*: a revision with new species. *Fungal Diversity* 55 (1): 77–108. https://doi.org/10.1007/s13225-012-0152-2
Sandoval-Denis, M., Gené, J., Sutton, D.A., Cano-Lira, J.F., de Hoog, G.S., Decock, C.A., Wiederhold, N.P. & Guarro, J. (2016) Redefining *Microascus*, *Scopulariopsis* and allied genera. *Persoonia* 36 (1): 1–36. https://doi.org/10.3767/003158516X688027
Santiago, A.L.C.M.A. & Cavalcanti, M.A.Q. (2007) *Gilbertella persicaria* (Mucorales): a new record from Brazil. *Mycotaxon* 30 (102): 333–337.
Saumell, C.A., Padilha, T. & Santos, C. (2000) Nematophagous fungi in sheep faeces in Minas Gerais, Brazil. *Mycologial Research* 104 (8): 1005–1008. https://doi.org/10.1017/S095375629900218X
Saumell, C.A., Padilha, T., Santos, C. & Roque, M.V. (1999) Nematophagous fungi in fresh feces of cattle in the Mata region of Minas Gerais state, Brazil. *Veterinary Parasitolology* 82 (3): 217–220. https://doi.org/10.1016/S0304-4017(99)00022-9
Saumell, C.A. & Padilha, T. (2000) Influence of weather and time of deposition on sheep faeces colonization by nematophagous fungi in the Mata region of Minas Gerais State. *Brazilian Applied Soil Ecology* 14: 63–70. https://doi.org/10.1016/S0929-1393(99)00045-1
Santiago, A.L.C.M.A., Trufem, S.F.B., Malosso, E., Santos, P.J.P. & Cavalcanti, M.A.Q. (2011) Zygomycetes from herbivore dung in the ecological reserve of Dois Irmãos, Northeast Brazil. *Brazilian Journal of Microbiology* 42 (1): 89–95. https://doi.org/10.1590/S1517-83822011000100012
Schipper, M.A.A. (1973) A study on variability in *Mucor hiemalis* and related species. *Studies in Mycology (Baarn)* 4: 1–40.
Schipper, M.A.A. (1975) *Mucor mucedo*, *Mucor flavus* and related species. *Studies in Mycology (Baarn)* 10: 1–33.
Schipper, M.A.A. (1978) On certain species of *Mucor* with a key to all accepted species. *Studies in Mycology (Baarn)* 17: 1–52.
Schumacher, T. (1990) *The genus Scutellinia (Pyronemataceae).* Copenhagen, Council for Nordic Publ. in Botany.
Seifert, K.A., Kendrick B. & Murase G. (1983) *A Key to Hyphomycetes on Dung*. Ontario, Department of Biology, University of Waterloo.
Seifert, K.A., Morgan-Jones, G., Gams, W. & Kendrick, B. (2011) *The genera of Hyphomycetes*. Utrecht, CBS-KNAW Fungal Biodiversity Centre.
Senanayake, I. (2016) Phomatosporales ord. nov. and Phomatosporaceae fam. nov., to accommodate *Lanspora*, *Phomatospora* and *Tenuimurus*, gen. nov. *Mycosphere* 7 (5): 628–641. https://doi.org/10.5943/mycosphere/7/5/8
Seth, H.K. (1971) The genus *Lophotrichus* Benjamin. *Nova Hedwigia* 19: 591–599.
Silva, P.S., Cortez, V.G. & Silveira, R.M.B. (2008) Strophariaceae (Agaricales, Basidiomycota) no Parque Estadual de Itapuã, Viamão, Rio Grande do Sul: chave para identificação das espécies. *Revista Brasileira de Biociências* 6 (3): 253–258.
Singer, R. (1986) *The Agaricales in modern taxonomy.* Koenigstein, Koeltz Scientific Books.
Smith, N.S. (2007) An Overview of Ecological and Habitat Aspects in the Genus *Fusarium* with Special Emphasis on the Soil-Borne Pathogenic Forms. *Plant Pathology Bulletin* 16: 97–120.
Souza, C.A.F. de, Lima, D.X., Gurgel, L.M.S. & Santiago, A.L.C.M.A. (2017) Coprophilous Mucorales (Ex Zygomycota) from three areas in the semi-arid of Pernambuco, Brazil. *Brazilian Journal of Microbiology* 48 (1): 79–86.

https://doi.org/10.1016/j.bjm.2016.09.008
Stchigel, A.M., Cano, J.F., Abdullah S.K. & Guarro, J. (2004) New and interesting species of *Monascus* from soil, with a key to the known species. *Studies in Mycology* 50: 299–306.
Subramanian, C.V. & Lodha, B.C. (1968) Two interesting coprophilous fungi from India. *Current Science* 37: 245–248.
Summerbell, R.C., Gueidan, C., Schroers, H.-J., de Hoog, G.S., Starink, M., Rosete, Y.A., Guarro, J. & Scott, J.A. (2011) *Acremonium* phylogenetic overview and revision of *Gliomastix*, *Sarocladium*, and *Trichothecium*. *Studies in Mycology* 68: 139–162. https://doi.org/10.3114/sim.2011.68.06
Tretter, E.D., Johnson, E.M., Benny, G.L., Lichtwardt, R.W., Wang, Y., Kandel, P., Novak, S.J., Smith, J.F. & White, M.M. (2014) An eight-gene molecular phylogeny of the Kickxellomycotina, including the first phylogenetic placement of Asellariales. *Mycologia* 106 (5): 912–935.
https://doi.org/10.3852/13-253
Tulloch, M. (1972) The genus *Myrothecium*. *Mycological Papers* 130: 1–42.
Uljé, K. & Bas, C. (1992) Studies in *Coprinus* II. Subsection *Setulosi* of section *Pseudocoprinus*. *Persoonia* 14: 275–339.
Uljé, K. & Noordeloos, M.E. (1999) Studies in *Coprinus* V-*Coprinus* Section *Coprinus*, Revision of subsection Lanatuli Sing. *Persoonia* 17: 165–199.
Upadhyay, H.P. (1981) *A Monograph of Ceratocystis and Ceratocystiopsis*. Athens, University of Georgia Press.
Vakili, N.G. (1989) *Gonatobotrys simplex* and its teleomorph, *Melanospora damnosa*. *Mycologial Research* 93 (1): 67–74.
https://doi.org/10.1016/S0953-7562(89)80139-X
Viriato, A. (2008) *Pilobolus* species found on the herbivore dung from the São Paulo Zoological Park, Brazil. *Acta Botanica Brasilica* 22 (3): 614–620.
https://doi.org/10.1590/S0102-33062008000300002
Visagie, C.M., Hirooka, Y., Tanney, J.B., Whitfield, E., Mwange, K., Meijer, M., Amend, A.S., Seifert, K.A. & Samson, R.A. (2014a) *Aspergillus*, *Penicillium* and *Talaromyces* isolated from house dust samples collected around the world. *Studies in Mycology* 78: 63–139.
https://doi.org/10.1016/j.simyco.2014.07.002
Visagie, C.M., Houbraken, J., Frisvad, J.C., Hong, S.-B., Klaassen, C.H.W., Perrone, G., Seifert, K.A., Varga, J., Yaguchi, T. & Samson, R.A. (2014b) Identification and nomenclature of the genus *Penicillium*. *Studies in Mycology* 78: 343–371.
https://doi.org/10.1016/j.simyco.2014.09.001
Wang, C.Y., Mo, M.H., Li, X., Tian, B.Y. & Zhang, K.Q. (2007) Morphological characteristics and infection processes of nematophagous *Harposporium* with reference to two new species. *Fungal Diversity* 26: 287–304.
Weber, R.W.S. & Webster, J. (1997) The coprophilous fungus *Sphaeronaemella fimicola*: a facultative mycoparasite. *Mycologist* 11: 50–51.
https://doi.org/10.1016/S0269-915X(97)80032-8
Weber, R.W.S. & Webster, J. (1998) Stimulation of growth and reproduction of *Sphaeronaemella fimicola* by other coprophilous fungi. *Mycological Research* 102: 1055–1061.
https://doi.org/10.1017/S0953756297006011
Yilmaz, N., Visagie, C.M., Houbraken, J., Frisvad, J.C. & Samson, R.A. (2014) Polyphasic taxonomy of the genus *Talaromyces*. *Studies in Mycology* 78: 175–341.
https://doi.org/10.1016/j.simyco.2014.08.001
Zheng, R. & Chen, G.Q. (2001) A monograph of *Cunninghamella*. *Mycotaxon* 80: 1–75.
Zheng R.-Y., Chen G.-Q., Huang, H. & Liu X.-Y. (2007) A monograph of *Rhizopus*. *Sydowia* 59: 273–372.
Zheng, R., Liu, X. & Wang, Y. (2017) *Circinella* (Mucorales, Mucoromycotina) from China. *Mycotaxon* 132 (1): 43–62.
https://doi.org/10.5248/132.43