



PERSOONIAL Reflections

Editorial: Celebrating 50 years of Fungal Biodiversity Research

The year 2009 represents the 50th anniversary of *Persoonia* as an international journal of mycology. Since 2008, *Persoonia* is a full-colour, *Open Access* journal, and from 2009 onwards, will also appear in PubMed, which we believe will give our authors even more exposure than that presently achieved via the two independent online websites, www.IngentaConnect.com, and www.persoonia.org. The enclosed free poster depicts the 50 most beautiful fungi published throughout the year. We hope that the poster acts as further encouragement for students and mycologists to describe and help protect our planet's fungal biodiversity. As 2010 is the international year of biodiversity, we urge you to prominently display this poster, and help distribute

the message that without fungi as basal link in the food chain, there will be no biodiversity at all.

May the *Fungi* be with you!

Editors-in-Chief:

Prof. dr PW Crous

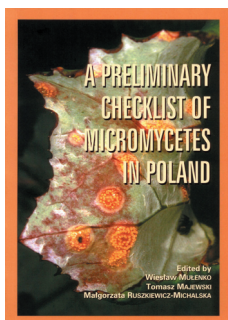
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Dr ME Noordeloos

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Book Reviews

Muilenko W, Majewski T, Ruskiewicz-Michalska M (eds). 2008. *A preliminary checklist of micromycetes in Poland*. Biodiversity of Poland, Vol. 9. Pp. 752; soft cover. Price 74 €. W. Szafer Institute of Botany, Polish Academy of Sciences, Lubicz, Kraków, Poland. ISBN: 978-83-89648-75-4.

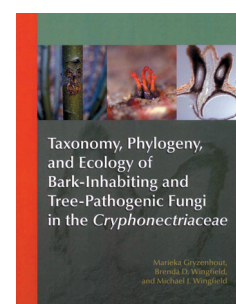


The present book forms part of the series depicting the Biodiversity of Poland. To date four published volumes of the series have been devoted to fungi, namely *Basidiomycetes*, *Ascomycetes*, lichens and lichenicolous fungi, and slime moulds. The present book covers the *Ascomycota*, *Basidiomycota*, *Chytridiomycota*, *Glomeromycota*, *Hypochytridiomycota*, *Oomycota*, *Plasmodiophoromycota*, *Zygomycota*, as well as anamorphic fungi which belong mostly to the *Ascomycota* (micromycetes), with the latter category representing the larger part of the book. In general the classification follows the latest scheme, with the various fungal orders linked to different authors. Taxa are well documented, and supported by the substrate on which they occur, numerous references, and in many cases nomenclatural synonymies and other relevant data. This represents an impressive book, which is far more than just a checklist of fungal taxa and their hosts, and will make a valuable contribution to all mycological libraries.

PW Crous

Gryzenhout M, Wingfield BD, Wingfield MJ. 2009. *Taxonomy, phylogeny, and ecology of bark-inhabiting and tree-pathogenic fungi in the Cryphonectriaceae*. Pp. 136; soft cover, 14 colour and 38 black & white plates, 16 line drawings. Price: \$119.00. APS Press, 3340 Pilot Knob Road, St. Paul, MN 55121. ISBN: 978-0-89054-367-2.

The *Cryphonectriaceae* include some of the most important tree pathogens in the world. Over the years I have personally helped collect populations of some species in Africa and South America, and have witnessed the devastation that these pathogens can bestow on trees and plantations. As such it is wonderful to finally have a treatment that summarises all mycological information known about the topic to date. The book has a section treating the history of the various taxonomic genera, as well as a section covering the diseases and their ecology. Of interest to many would be the sections on pathogen identification, as well as the dichotomous and synoptic keys. The *Cryphonectria* complex was the PhD topic of the first author, and the notes supporting each species in the book shows that she has an intimate knowledge of all species treated. The incredible and almost fanatical ability of MJW to recollect all these taxa from all corners of the world should also be acknowledged, as without such collections new hypotheses and significant progress would not be possible. The thorough approach, and detailed illustrations and descriptions, supported by numerous cultures and specimens in publicly accessible collections, sequences in GenBank, alignments in TreeBASE, and nomenclature in MycoBank, make this a gold standard for such monographic works. The authors need to be congratulated for many years of hard work, as they have once again moved the goalposts in a new direction. Of course every book leaves room for improvement. In spite of all the appendixes and indexes, I would have appreciated a strictly alphabetical treatment of taxa. I personally hate reading instruction manuals and indexes, and simply want to find things where they should be: alphabetical. The anamorphs and teleomorphs are all named, which is superfluous in this day and age, as one



name per genus is more than enough. The complex is split into 10 genera, but these are not supported by LSU data, so it remains to be seen if these genera survive the test of time. The book itself is beautiful, but the printer did not do justice

to some black and white plates, as I have seen the originals. In conclusion, if you like trees and tree diseases, you should order this book – it's a jewel, and will be the benchmark for many years to come.

PW Crous

Dr Machiel Noordeloos receives the Clusius Award

Dr. Machiel E. Noordeloos, mycologist at the Leiden University branch of the National Herbarium of the Netherlands, received the prestigious Clusius Award of the Hungarian Mycological Society on October 22, 2009, during the 27th International Cortinarius Conference, a six-day scientific meeting held in Nyíregyháza, Hungary, organized by the Hungarian Mycological Society.



Dr M.E. Noordeloos receives award from Prof. Erzsébet Jakucs

The medal and the certificate, named after Carolus Clusius, a pioneering botanist and mycologist of the 16th and early 17th century, is awarded on special occasions to an individual who has made an outstanding contribution to fungal taxonomy, marking a distinguished career in mycology. The award was presented to Machiel Noordeloos by Prof. Imre Rimóczi and Prof. Erzsébet Jakucs (President of the Hungarian Mycological Association), who praised his internationally renowned mycological work, and in particular his efforts to make his work accessible to a broad audience.

Machiel Noordeloos is one of the leading experts on fungal taxonomy, particularly that of the *Agaricales* and *Boletales* in Europe. He is the world authority on *Entoloma*, a speciose and morphologically and ecologically highly diverse genus. Noordeloos has been editor of the long-term Flora Agaricina Neerlandica project since 1990, a critical flora in 11 volumes of all 2200 species occurring in Western Europe. In addition, he is currently involved in monographic studies on the *Entolomataceae* in Europe, North America, Australia, and Malesia.



Clusius Medal.

Carolus Clusius was not only the 'Erasmus of the Botanical Sciences' but he is also regarded as the founder of mycology in connection with his pioneer mycological work on mushrooms in Central Europe. It is particularly amusing that, with Machiel Noordeloos, the award now "comes home" to Leiden, where Clusius founded the Hortus botanicus, became the first professor of Botany, and spent the last sixteen years of his life.

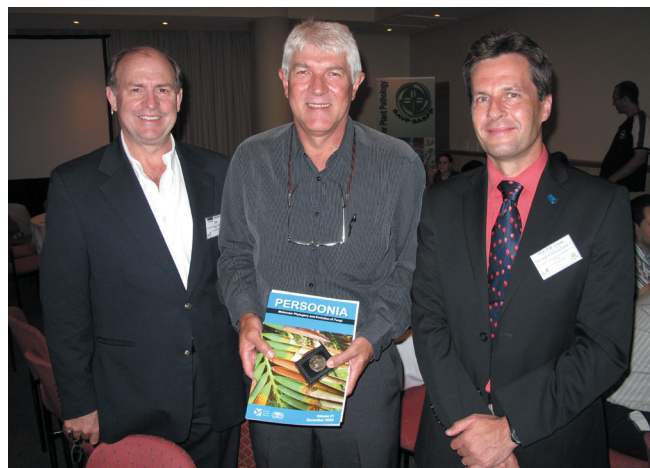
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Prof. dr Z.A. Pretorius awarded the Christiaan Hendrik Persoon Gold Medal by the Southern African Society for Plant Pathology (SASPP)

In January 2009 Prof. dr Z.A. (Sakkie) Pretorius (Professor and Chairperson of the Department of Plant Sciences, University of the Free State, Bloemfontein, South Africa) was awarded the Persoon Gold Medal by the SASPP, in recognition for outstanding achievement in Plant Pathology.

Prof. Pretorius has been professionally involved with plant pathology for over 30 years since his appointment at the ARC - Small Grain Institute at Bethlehem in 1978. Among his many achievements in this field have been the first comprehensive and ongoing characterisation of pathogenic variation in the wheat leaf rust fungus in South Africa, identification of germplasm with effective genetic resistance (with emphasis on field resistance) to local leaf and stem rust pathotypes and the establishment of evaluation and selection protocols. By means of backcrossing, he has successfully introduced eight leaf rust resistance genes into well adapted wheat cultivars for use in larger breeding programmes in South Africa and has also identified adult-plant leaf rust resistance in wild relatives of wheat. As a result of his



From left to right: professors M.J. Wingfield, Z.A. Pretorius (current recipient of award), and P.W. Crous during the award ceremony.

inputs, more directed selection or breeding for rust resistance is now possible in South Africa. The economic impact of this work is impossible to calculate but has resulted in savings of many millions of South African Rands for small grain farmers which can be illustrated by the fact that a reduction of only one spray in rust-prone areas results in a total financial saving of approximately ZAR24 million per season. Prof. Pretorius has also made major contributions to our understanding of other devastating diseases such as *Puccinia striiformis* on wheat, *P. helianthi* on sunflower, *P. coronata* and *P. graminis* on oat, *P. hordei* on barley, and *P. sorghi* on maize.

He has played an essential role in providing continuity in rust pathology in Southern Africa, and his scientific contributions have been widely recognised, as can be seen from the numerous awards he has received to date. It is thus fitting that the SASPP bestowed on him their highest honour, namely the Christiaan Hendrik Persoon Gold Medal, in recognition for a career of excellence in fungal research.

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Update on Fungal DNA Barcoding Campaigns

Network for the Indoor Mycobiota Barcode of Life (IM-BOL)

About 200 culturable mould species are common in buildings in North America and Europe. The uncultured profile of indoor moulds is poorly understood. Recent studies show that uncultured fungi occur in great abundance, but their significance and frequencies remain unknown. We assembled a set of dust samples from all continents except the Antarctic, and subjected them to standard protocols for isolation and morphological identification, high throughput dilution to extinction, and to 454 pyrosequencing. The internal transcribed spacer of the ribosomal operon (ITS) was used as a standard barcoding marker in all work, but we are also mining available fungal genomes searching for barcode markers with higher resolution. About 1000 cultures have been isolated and identified using conventional techniques. The high-throughput method was adapted for house dust and implemented on the global sample, and the first set of 600 cultures is now being sequenced. First pyrosequencing results demonstrate a taxonomically diverse and geographically patterned mycobiota, with diversity higher in temperate zones than in the tropics; a subset of 36 taxa (Dothideomycetes) has a cosmopolitan distribution, with regional fungal profiles exhibiting spatial autocorrelation at national and hemispheric scales.

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Quarantine Barcoding of Life (QBOL)

QBOL, a project funded by the European Commission Framework 7 Programme, is generating barcode sequences from a set of genetic regions for a selected set of quarantine organisms: arthropods, bacteria, fungi, nematodes, phytoplasmids and viruses. The research on these organisms is carried out in six different work packages and more information on the whole project is available from its website, www.qbol.org. QBOL follows the European Union Directive list with all the regulated Q-organisms and the EPPO (European and Mediterranean Plant Protection Organization) list where 90 % of all Q-organisms are listed (A1-list with organisms that do not exist in the EU, and the A2-list of organisms that are present but controlled in Europe). The research coordinators represent organizations with a huge expertise on taxonomy as well as on sequencing activities regarding the taxa involved.

The following genera were selected from the EU Directive 2000/29/EC (8 May 2000) and EPPO (September 2008) lists for quarantine organisms important to the European Union:

- *Ceratocystis* (*C. fagacearum*; *C. fimbriata* f. sp. *platani*; *C. virescens*)
- *Melampsora* (*M. farlowii*; *M. medusae*)
- *Monilinia* (*M. fructicola*)
- *Mycosphaerella* (*M. dearnessii*; *M. gibsonii*; *M. laricis-leptolepidis*; *D. populorum*; *Ph. angolensis*; *S. lycopersici* var. *mailagutii*)
- *Puccinia* (*P. pittieriana*)
- *Thecaphora* (*T. solani*)

These selections were based on the high level of expertise present in each of the partners involved, as well the availability of starting material and related species for comparison. To have a good and efficient barcode it is very important to also have closely related species so that it is possible to identify the most effective part of a sequence that can serve as the barcode region. Other important genera (*Alternaria*, *Colletotrichum*, *Guignardia*, *Phytophthora* and *Phoma*) are already being addressed in other programmes (mainly in the UK and the Netherlands) and are therefore not included in the QBOL project. However, QBOL has an agreement with these programmes that their data will be made available to the QBOL fungal database. The 'Barcoding Fungi' work package aims to obtain effective barcode regions that can be used to accurately identify the species listed above. To achieve this, the work package aims to target at least 15 individuals (preferably from different localities) for each quarantine species and at least two individuals from three or more close relatives.

One of the more crucial parts of the project for the group is to identify a gene(s) that can be used as a reliable barcode region(s). The Internal Transcribed Spacer (ITS) regions of the nuclear ribosomal RNA genes are commonly used for fungi, but lacks barcoding resolution in some genera. For many fungi, the general tendency therefore is to use ITS as a starting marker to identify to genus or species-complex level, and then to apply an additional gene(s) to identify the species in question. A screening process is currently underway to evaluate the variation for various loci between individuals of the same species and also between different species. The hope is to find a single region suitable for the identification of all these species, but a worst-case scenario can also be envisaged where a system might need to be implemented where two regions are used. All of the reference data generated for the project will be placed in the online version of the QBOL database. If you have material that could contribute to or expand the fungal database, please contact the co-ordinator of the Barcoding Fungi work package: Ewald Groenewald (e.groenewald@cbs.knaw.nl).

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Ulocladium populi E.G. Simmons, G. Newcombe & A. Shipunov, *sp. nov.*

Conidia in cultura praecoci (1–2 d) conspicue longe ovoidea vel longe ellipsoidea, ad $30\text{--}40 \times 8\text{--}12\ \mu\text{m}$, 3–6 transverse septata et 1–4 longiseptata. Conidia alia abunda, obovoidea vel sphaeroidea, ad $12\text{--}18 \times 10\text{--}15\ \mu\text{m}$, 1–3 transverse septata et 1–2 longe vel oblique septata. Conidiophora plerumque $25\text{--}50 \times 4\text{--}5\ \mu\text{m}$, 6–8 geniculata et conidiogena; interdum ad $150\ \mu\text{m}$ longa, in maturitate multigeniculata et 15–20 conidiogena.

Etymology. Named after its host, *Populus trichocarpa*.

Conidium production begins readily and is abundant within the first 1–2 d of hyphal development in potato-carrot agar (PCA)¹ plates. The most conspicuous sporulation elements in young growth are alternarioid; they originate with a typical apiculate ulocladioid base but become long-ovoid to long-ellipsoid and remain so in a large proportion as they mature. Unlike other *Ulocladium* taxa with multiplex conidium morphology described¹ alternarioid conidia of *U. populi* constitute a high percentage of the 5–7 d sporulation population. Conspicuously obovoid and sphaeroid conidia that develop with a narrow, typically ulocladioid base are equally abundant but less conspicuous in 1–2 d growth. In older colonies these ulocladioid conidia dominate the population without obscuring the abundant alternarioid elements. Sporulation is extremely abundant on a turf of erect or inclined conidiophores which, however, are individually distinguishable at $50 \times$ magn., each with an apical cluster of conidia. *Most conidiophores* are short, $25\text{--}50 \times 4\text{--}5\ \mu\text{m}$. Each bears a solitary terminal conidium. Subsequently the conidiophore apex develops into a series of 6–8 short, consecutive, closely sympodial extensions, each with a solitary conidium. Longer conidiophores also arise at the agar surface. These are abundant but not dominant within the colony and are interspersed among the far more abundant short conidiophores. The longer conidiophores usually are branched, with each branch developing its own sympodial apical region of conidiogenous cells. The longer conidiophore complex is up to $150\ \mu\text{m}$ long, commonly with a terminal cluster of 15–20 conidia. *Long-ovoid and ellipsoid conidia* become $30\text{--}40 \times 8\text{--}12\ \mu\text{m}$ with 3–6 transverse septa and a single longiseptum in each of 1–4 of the transverse segments; these alternarioid spores are smooth, rarely becoming inconspicuously ornamented in age. Obovoid and sphaeroid conidia become $12\text{--}18 \times 10\text{--}15\ \mu\text{m}$ with 1–3 transverse septa and 1 longitudinal or oblique septum in 1–2 of the transverse segments; the outer wall of these typically ulocladioid spores develop a densely pustulate ornamentation that often obscures internal septation in mature conidia. Conidium colour becomes a dilute dull brown, with ulocladioid conidia becoming a darker brown as ornamentation matures. Nearly all conidia remain

solitary. Within the extremely large population of conidia only a rare individual generates an apical or lateral short conidiophore with 1–4 conidiogenous sites. The *U. populi* colony on PCA at 5–6 d under a daily light/dark cycle of 8/16 h is c. 5 cm diam with 5 pairs of concentric rings of growth and sporulation. The colony is totally conidial, in that all surface hyphae produce abundant conidiophores; non-sporulating aerial hyphae are not present. Closest species is *U. atrum*.

Typus. USA, Idaho, in the stem of *Populus trichocarpa*; *holotypus*: CBS H-20385, a dried culture preparation ex E.G.S. 53.001 = CBS 123360, from George Newcombe isolate no. CIDU1, Lapwai Canyon, Idaho, USA, March 2, 2005. GenBank EF589900 and EF589845, MycoBank MB515393.

Notes — Following extensive sampling of endophytic isolates of *Ulocladium* and *Alternaria*² a phylogenetic tree was constructed that demonstrates the affinities of *U. populi* to a group of isolates and taxa associated with *U. atrum*. Our analyses support the monophyly of the *U. atrum* group. As in previous analyses³, a clade comprised of *U. chartarum*, *Alternaria cheiranthi*, and *Embellisia indefessa* was sister to the *U. atrum* complex. However, two isolates identified on the basis of morphology as representative of *U. atrum* were phylogenetically distinct: ATCC 18040 (AF229486, AY563318) shown as *U. atrum*, and EGS 30-188 that is identical to haplotype '068g'. Also problematic from a taxonomic point of view is the identity of the ex-type culture of *U. dauci* with one of the representative cultures of *U. atrum*, EGS 30-188. Apart from *U. populi*, described above, the resolution of other members of the complex appears to be poor. The paraphyly of *Alternaria* is clearly seen in the tree. There were at least four different clades. One comprised all *Lewia* (*Alternaria* teleomorph) sequences, *A. oregonensis*, *A. photistica*, and 12 endophytic haplotypes of *Alternaria* isolated from *Centaurea stoebe*. The second comprised *A. alternata*, *A. arborescens*, *A. tenuissima*, *A. longipes* and 15 of the endophytic haplotypes of *Alternaria* isolated from *C. stoebe*. The third (*A. crassa* and others) and fourth (*A. carotiincultae* and others) clades did not include endophytes obtained from *C. stoebe*. The '674n' haplotype from *C. stoebe* (close to *Alternaria malorum* sequences from GenBank) did not ally with the first *Alternaria* clade in MP trees (not shown); even on ML trees its branch was quite long. None of the 27 endophytic haplotypes of *Alternaria* and *Lewia* belonged to the *U. atrum* complex.

For phylogenetic tree and line drawing see MycoBank MB515393.

Colour illustrations. *Populus trichocarpa*, in Lapwai Canyon, Idaho, USA; *Ulocladium populi*, portion of conidiophore and ornamentation of conidia from different angles. Scale bars = $10\ \mu\text{m}$.

References. ¹Simmons EG. 1998. Multiplex conidium morphology in species of the *Ulocladium atrum* group. Canadian Journal of Botany 76: 1533–1539. ²Shipunov A, Newcombe G, Raghavendra AKH, Anderson CL. 2008. Hidden diversity of endophytic fungi in an invasive plant. American Journal of Botany 95: 1096–1108. ³Hong SG, Cramer R, Lawrence CB, Pryor BM. 2005. Alt a 1 allergen homologs from *Alternaria* and related taxa: analysis of phylogenetic content and secondary structure. Fungal Genetics and Biology 42: 119–129.

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Clavispora reshetovae

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***Clavispora reshetovae* A. Yurkov, A.M. Schäfer & Begerow, sp. nov.**

Cultura in striis in agarum cum dextroso et peptono et extracto levidinis (GPY) post unum mensem candida ad crema, obscura, laevis, margine integro. In agarum cum extracto malti post 3–5 dies ad 25 °C cellulae sphaeroideae ad ovoideae ($1.5\text{--}2.5 \times 2.5\text{--}5.0\ \mu\text{m}$), singulae, binae vel aggregatae, multilateraliter gemmantes. Post hebdomades tres in agarum cum extractis levidinis et malti (YM) vel in agarum cum extracto Solani tuberosi et dextroso (PD) ad 20–25 °C pseudohyphae vel hyphae verae desunt. Status teleomorphicus post cultivationem hebdomadam tres ad 16 °C in agarum cum extracto Solani tuberosi et dextroso (PD) observatus, homothallicus. Asci conjugatione cellularum vel conjugatione cellularum gemmarumque oriuntur, ascosporis duabus. Glucosum fermentatur. Glucosum, D-galactosum, L-sorbose, D-xylose, L-rhamnosum (exiguum, lente), sucrosus, D-maltosus, a,a-trehalosus, cellobiosus, salicin, glycerolum, ribitolum (lente), D-glucitol, D-mannitol, ethanol, acidum succinicum (exiguum, lente), acidum DL-lacticum (exiguum, lente) et acidum citricum (exiguum, lente) assimilantur at non D-glucosaminum, L-arabinosum, D-arabinosum, D-ribosum, lactosum, raffinose, melezitose, inulinum, amyllum solubile, erythritolum, galactitol, nec inositolum. Assimilatio ethylamini et lysini ad non kalii nitrati nec kalii nitrosi. Materia amyloidea non formatur. Ureum non finditur. Vitamina externa ad crescendum necessaria. Temperatura maxima crescentiae: 30 °C.

Etymology. The specific epithet '*reshetovae*' is in honour of the Russian microbiologist Irina S. Reshetova for her contributions to the studies of soil yeasts.

On Glucose Peptone Yeast extract Agar (GPYA), after 1 mo at 25 °C, the streak culture is white to cream, dull and smooth. The margin is entire. After growth on malt extract agar for 3–5 d at 25 °C, cells are spheroid to ovoid ($1.5\text{--}2.5 \times 2.5\text{--}5.0\ \mu\text{m}$), occurring singly, in pairs or in small clusters and proliferating by multilateral budding. Pseudohyphae and true hyphae are not observed after 3 wk in plate culture on Yeast extract Malt extract malt (YM) and potato-dextrose agar (PDA, Difco) at 20–25 °C.

Teleomorphic stage was obtained on PDA after incubation for 3 wk at 16 °C. Ascospores were also observed on GPYA after replating from PDA. Ascus formation may be preceded by either conjugation between independent cells or by conjugation between a parent cell and a bud. Asci contain two ovoid ascospores with a small ring, and after maturation, ascospores are liberated from the ascus and tend to agglutinate.

Glucose is fermented. Assimilation of carbon compounds: D-galactose, L-sorbose, D-xylose, L-rhamnose (weak), sucrose,

D-maltose, a,a-trehalose, cellobiose, salicin, glycerol, ribitol (delayed), D-glucitol, D-mannitol, ethanol, DL-lactate (weak), succinate (weak) and citric acid (weak). No growth occurs on D-glucosamine, L-arabinose, D-arabinose, lactose, raffinose, melezitose, inulin, soluble starch, erythritol, galactiol, and myo-inositol. Assimilation of nitrogen compounds: potassium nitrate and nitrite (negative), L-lysine and ethylamine (positive). Starch-like compounds are not produced. Urease activity is negative. Growth on vitamin-free medium is negative. Maximal growth temperature: 30 °C.

Typus. GERMANY, Thuringia, National Park Hainich, soil collected at the grassland, 51.2239 N, 10.3807 E, April 2008, HEG-9-BEB; holotype, culture ex-type HEG-9-2 = CBS 11556, A. Yurkov, GenBank FN433523 (ITS-region), FN428961 (D1/D2 domain of 26S rRNA gene), FN433522 (16S rRNA gene), MycoBank MB515101.

Notes — During a project aimed to study yeasts in soils under different land use, several cultures representing a novel teleomorphic yeast were isolated by plating soil suspensions on a solid acidified GPYA medium. Physiological analysis and phylogenetic placement showed this species to be related to the ascomycetous genus *Clavispora* (1979), *Metschnikowiaceae*^{1,2}. The novel species belongs to the cluster with two known *Clavispora* spp., namely *C. lusitaniae* and *C. opuntiae*, and several anamorphic *Candida* spp. *Clavispora lusitaniae* occurs in different substrates of plant and animal origin, whereas *C. opuntiae* is assumed to be associated with necrotic cacti². Although there is no strong support for this clade, we propose the novel species to be described in the genus *Clavispora*, as it resembles many of the properties and does not contradict the current diagnosis of the genus.

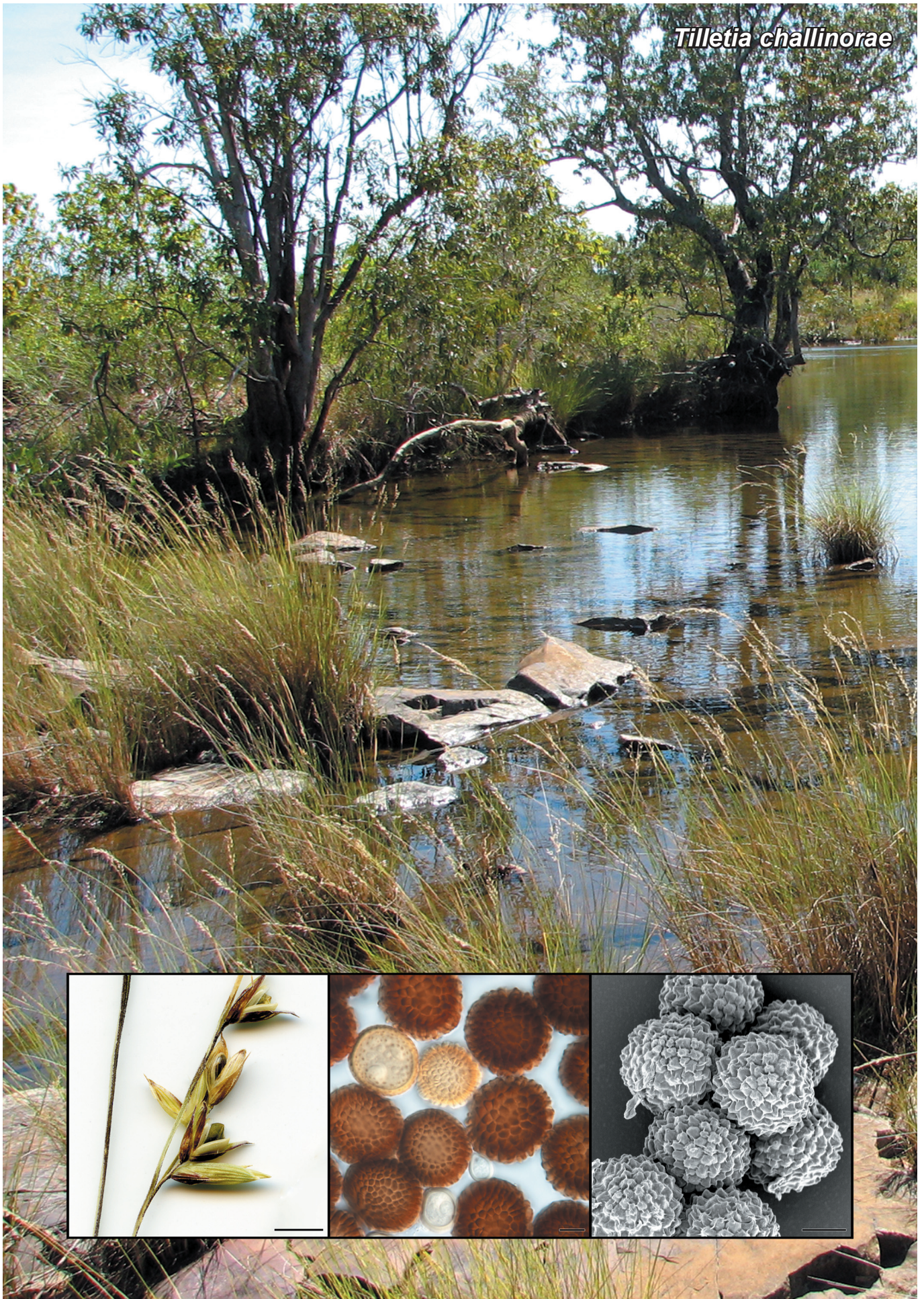
For phylogenetic tree see MycoBank MB515101.

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Colour illustrations. Grassland in the National Park Hainich; soil core that yielded the ex-type strain; phase contrast micrographs of *Clavispora reshetovae*: vegetative cells, isogamous conjugation, pedogamous asci, asci of different origin and liberated ascospores. Scale bars = 10 μm .

References. ¹Rodrigues de Miranda L. 1979. *Clavispora*, a new yeast genus of the Saccharomycetales. *Antonie van Leeuwenhoek* 45: 479–483. ²Lachance MA, Daniel HM, Meyer W, Prasad GS, Gautam SP, Boundy-Mills K. 2003. The D1/D2 domain of the large-subunit rDNA of the yeast species *Clavispora lusitaniae* is unusually polymorphic. *FEMS Yeast Research* 4: 253–258.

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Tilletia challinorae

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***Tilletia challinorae* McTaggart & R.G. Shivas, sp. nov.**

Sporae globosae, subglobosae vel late ellipsoideae, $27\text{--}34 \times 25\text{--}29\text{ }\mu\text{m}$, altobrunneae, opaceae, verrucis acutatis, conicis, $3\text{--}4\text{ }\mu\text{m}$ altis et $3\text{ }\mu\text{m}$ latis, superficiali aspectu visae maculae irregulares, subpolyangulares, $10\text{--}15\text{ }\mu\text{m}$ per sporam diametro. Immaturae sporae globosae, cinnamomeae, $22\text{--}29 \times 21\text{--}27\text{ }\mu\text{m}$.

Etymology. Named after Victoria Louise Challinor, who discovered this and several other rare smut fungi.

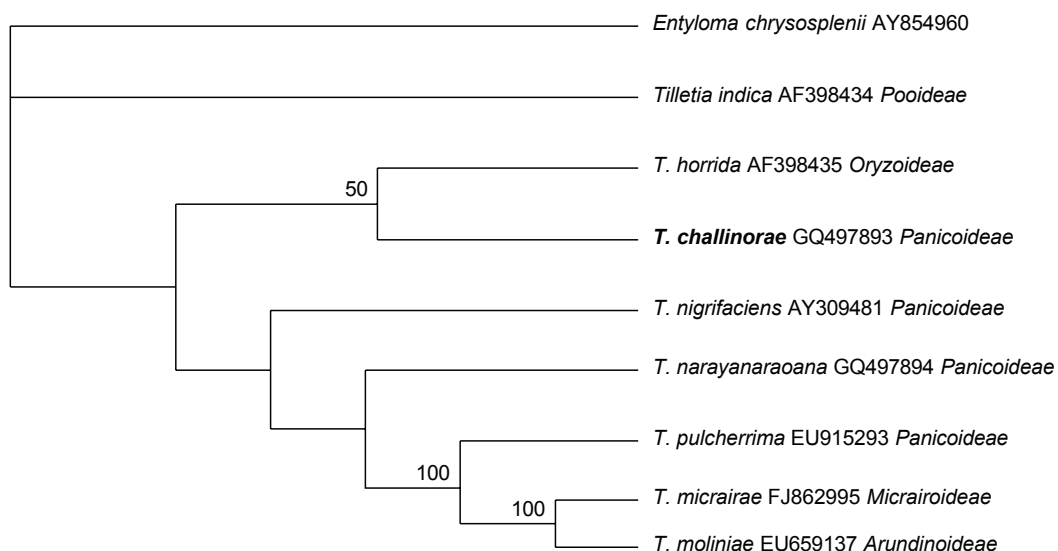
Sori in some slightly swollen ovaries of an inflorescence, protruding past the lemma, but shorter than the glumes, ovoid, $1.5\text{--}2.5 \times 1.0\text{ mm}$, covered by a peridium, initially green becoming dark brown, rupturing irregularly releasing the black, powdery spore mass mixed with sterile cells. **Spores** when mature globose, subglobose or broadly ellipsoidal, $27\text{--}34 \times 25\text{--}29\text{ }\mu\text{m}$, dark chestnut-brown, opaque, ornamented with acute conical warts, $3\text{--}4\text{ }\mu\text{m}$ high and $3\text{ }\mu\text{m}$ wide at base, $10\text{--}15$ per spore diam, in surface view appearing as irregular subpolyangular spots. Immature spores pale brown, globose, $22\text{--}29 \times 21\text{--}27\text{ }\mu\text{m}$. **Sterile cells** globose, subglobose to broadly ellipsoidal, $14\text{--}26 \times 13\text{--}23\text{ }\mu\text{m}$, hyaline to sub-hyaline, pale yellow when larger; wall c. $1.0\text{ }\mu\text{m}$ thick, smooth to finely verruculose.

Typus. AUSTRALIA, Western Australia, Mitchell Plateau, Surveyor's Pool, $14^\circ 40' 07''\text{ S}$, $125^\circ 44' 25''\text{ E}$, *Panicum trachyrachis*, 12 May 2009, V.L. Challinor, A.R. McTaggart, M.J. Ryley, C.E. Gambley, T. Scharaschkin, M.D.E. & R.G. Shivas, BRIP 52502, holotype, PERTH 07702639, isotype; ITS sequence GenBank GQ497893, MycoBank MB515157.

Notes — Thirty-eight species of *Tilletia* have been reported from Australia^{1,2,3} of which 14 are endemic to northern Australia. Worldwide, 12 species of *Tilletia* are known to occur on *Panicum*. One of these, *T. narayanaraoana* on *Panicum trachyrachis*, is known from Australia. *Tilletia challinorae* differs from *T. narayanaraoana* in having smaller sori, larger spores (*T. narayanaraoana* has spores $20\text{--}28 \times 16\text{--}25\text{ }\mu\text{m}$) with acute, single warts rather than blunt, agglutinated, filiform warts. Ten species of *Tilletia* occur as localised infections on *Panicum*. Of these, the most morphologically similar is *T. pulcherrima*, which has smaller spores ($20\text{--}29.5 \times 20\text{--}28\text{ }\mu\text{m}$) that also differ in colour, ornamentation and the presence of a mucilaginous sheath surrounding the spores.

BLASTn results of the ITS sequence of *T. challinorae* had high identity to sequences of *T. horrida* on *Oryza sativa* (AF398435.1, 87 % identical over 97 % query coverage), *T. pulcherrima* on *Panicum virgatum* (EU915293.1, 91 % identical over 70 % query coverage) and *T. narayanaraoana* on *Panicum trachyrachis* (GQ497894, 87 % identical, over 90 % query coverage). Genomic DNA and cloned ITS plasmid of *T. challinorae* (holotype) are stored in the Australian Biosecurity Bank (<http://www.padil.gov.au/pbt/>).

Acknowledgements ARM would like to acknowledge the support of the Cooperative Research Centre for National Plant Biosecurity.



Majority-rule consensus tree (TL = 445; CI = 0.775; RI = 0.552; RC = 0.428) obtained using parsimony in an exhaustive search from an ITS sequence alignment using PAUP v4.0b10. The bootstrap support values from 1 000 replicates are shown at the nodes. The species described here is printed in **bold face**. The tree was rooted to *Entyloma chrysosplenii* (GenBank AY854960). There is little statistical support for this tree, but it serves to show that *T. challinorae* is more closely related to species of *Tilletia* on grasses in the tribe *Panicoideae* than the tribe *Pooideae*, a host-pathogen relationship previously reported in a phylogenetic analysis of combined morphological and LSU sequence data⁴.

Colour illustrations. Creek at Surveyor's Pool, Mitchell Plateau, Western Australia; sori in ovaries of *Panicum trachyrachis*; mature (dark brown) and immature (pale brown) spores and sterile cells; spore wall seen in SEM. Scale bars (from left to right) = 2.5 mm, 10 μm , 10 μm .

References. ¹Ványi K, Shivas RG. 2008. Fungi of Australia: The smut fungi. ABRIS, Canberra; CSIRO Publishing, Melbourne. ²Shivas RG, McTaggart AR. 2009. Three new species of *Tilletia* on native grasses from northern Australia. Australasian Plant Pathology 38: 128–131. ³Barrett MD, Barrett RL, Shivas RG, McTaggart AR. 2009. *Tilletia micrairae*. Fungal Planet 33, Persoonia 22: 170–171. ⁴Castlebury LA, Carris LM, Ványi K. 2005. Phylogenetic analysis of *Tilletia* and allied genera in order Ustilaginales (Ustilaginomycetes; Exobasidiomycetidae) based on large subunit nuclear rDNA sequences. Mycologia 97: 888–900.

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***Macalpinomyces mackinlayi* McTaggart & R.G. Shivas, sp. nov.**

Sori in nonnullis ovaris inflorescentiae, longe cylindrici, $10\text{--}35 \times 1.0\text{--}1.5\ \mu\text{m}$, primo virides tum cinerei. Sporae globosae, subglobosae vel late ellipsoideae, $9\text{--}13 \times 8\text{--}12\ \mu\text{m}$, luteobrunneae; paries aequalis, dense opertus conicis spinis $1\text{--}2\ \mu\text{m}$ altis. Cellulae steriles in catervis irregularibus, cellulae singulae globosae, subglobosae, ellipsoideae, $5.5\text{--}10.0 \times 4.5\text{--}8.0\ \mu\text{m}$, hyalinae; paries aequalis, ca. $0.3\ \mu\text{m}$, levis.

Etymology. Derived from the host epithet.

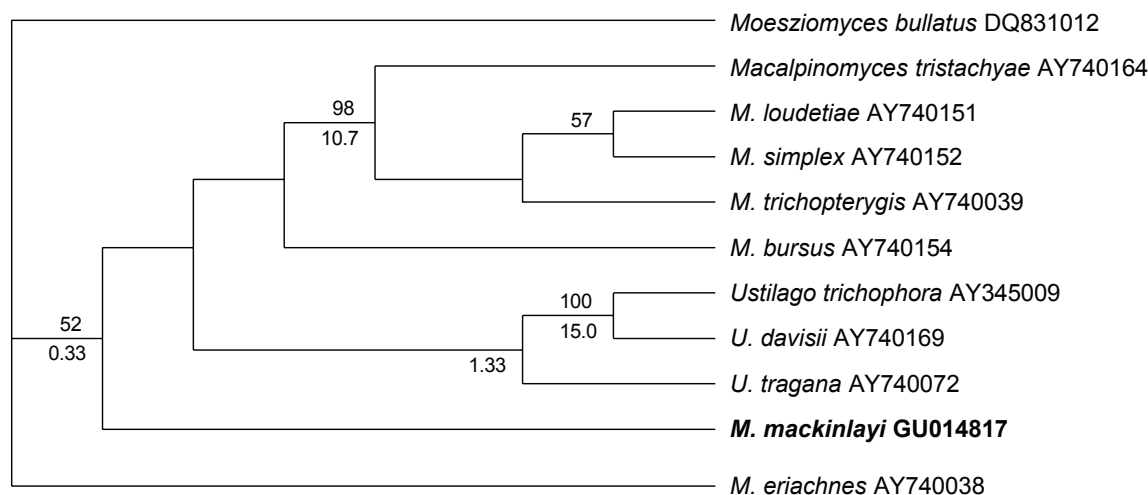
Sori in some ovaries of an inflorescence, hypertrophied, long-cylindrical, sometimes twisted, $10\text{--}35 \times 1.0\text{--}1.5\ \text{mm}$ wide, initially green becoming grey from the apex downwards, with reddish brown remnants about 2 mm long of the host pericarp at the apex, rupture longitudinally exposing the powdery spore mass mixed with sterile cells. Spores globose, subglobose or broadly ellipsoidal, $9\text{--}13 \times 8\text{--}12\ \mu\text{m}$, yellowish brown; wall even, densely covered in conical spines $1\text{--}2\ \mu\text{m}$ high. Sterile cells in large, loose, irregular groups; individual cells globose, subglobose, ellipsoidal or slightly irregular, $5.5\text{--}10.0 \times 4.5\text{--}8.0\ \mu\text{m}$, hyaline; wall even, c. $0.3\ \mu\text{m}$ thick, smooth.

Typus. AUSTRALIA, Western Australia, c. 35 km north of Drysdale River, alt. c. 380 m, $15^\circ 23' 13''$ S, $126^\circ 16' 58''$ E, *Eulalia mackinlayi*, 10 May 2009, A.R. McTaggart, V.L. Challinor, M.J. Ryley, C.E. Gambley, T. Scharaschkin, M.D.E & R.G. Shivas, BRIP 52549, holotype; ITS sequence GenBank GU014817, MycoBank MB515252. *Paratypus*, Western Australia, between King Edward River crossing and Mitchell Falls, 10 May 2009, A.R. McTaggart, V.L. Challinor, M.J. Ryley, C.E. Gambley, T. Scharaschkin, M.D.E & R.G. Shivas, BRIP 52546.

Notes — *Macalpinomyces* is a polyphyletic genus with many species referable to either *Ustilago* or *Sporisorium*¹. *Macal-*

pinomyces is represented in Australia by 12 taxa². *Macalpinomyces mackinlayi* is best placed in *Macalpinomyces* until the *Ustilago-Sporisorium-Macalpinomyces* genus complex is resolved. It lacks columellae, typically present in *Sporisorium* and has sterile cells, which are not a character of *Ustilago*. It is morphologically similar to other *Macalpinomyces* species that have sterile cells, hypertrophied sori derived from host material, and densely echinulate spores, e.g. *M. arundinella-setosae*, *M. tubiformis* and *M. siamensis*. *Macalpinomyces mackinlayi* occurs on *Eulalia mackinlayi*, which is only known from the Mitchell Plateau region in north-western Australia. Eight *Sporisorium* species have been recorded on *Eulalia*, seven of which destroy the entire inflorescence or all the spikelets in an inflorescence. *Sporisorium tripsicatum* has localised sori and can be distinguished from *M. mackinlayi* by the white sorus derived from fungal cells, the presence of spore balls and the verrucose rather than echinulate spores.

BLASTn results of the ITS sequence of *Macalpinomyces mackinlayi* (GU014817) had high identity to sequences of *M. tristachyae* on *Loudetiopsis chrysothrix* (GenBank: AY740164, 96 % identical over 90 % query coverage), *M. bursus* (as *Sporisorium bursum*) on *Themeda quadrivalvis* (GenBank: AY740154, 94 % identical over 90 % query coverage), *Ustilago trichophora* on *Echinochloa colona* (GenBank: AY345009, 94 % identical over 83 % query coverage) and *M. loudetiae* on *Loudetia flavida* (GenBank: AY740151, 91 % identical over 90 % query coverage). Genomic DNA of *M. mackinlayi* (holotype) is stored in the Australian Biosecurity Bank (<http://www.padil.gov.au/pbt/>).



Analysis of the ITS region of *Macalpinomyces mackinlayi* and some closely related taxa from GenBank in an exhaustive parsimony search using PAUP v4.0b4 yielded one tree (TL = 696; CI = 0.838; RI = 0.552; RC = 0.462). Bootstrap values from 1 000 replicates are shown above nodes and decay indices shown below nodes. The species described here is printed in bold face. The tree was rooted to *Moesziomyces bullatus* (Gen-

Bank DQ831012), a known outgroup of the *Ustilago-Sporisorium-Macalpinomyces* genus complex¹. A maximum likelihood analysis resolved a similar tree topology, except *M. mackinlayi* was sister to the *M. tristachyae* clade. This tree highlights that *Macalpinomyces* is a non-monophyletic group.

Acknowledgements ARM would like to acknowledge the support of the Cooperative Research Centre for National Plant Biosecurity.

References. ¹Stoll M, Begerow D, Oberwinkler F. 2005. Molecular phylogeny of *Ustilago*, *Sporisorium*, and related taxa based on combined analyses of rDNA sequences. *Mycological Research* 109: 342–356. ²Ványi K, Shivas RG. 2008. *Fungi of Australia: The Smut Fungi*. ABRIS, Canberra; CSIRO Publishing, Melbourne.

Colour illustrations. Drysdale River, Western Australia; sori in ovaries of *Eulalia mackinlayi*; spores and sterile cells; spore wall and sterile cells seen in SEM. Scale bars (from top to bottom) = 1 cm, 10 μm , 5 μm .

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Barnettozyma vustinii

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***Barnettozyma vustinii* A. Yurkov, A.M. Schäfer & Begerow, sp. nov.**

Cultura in striis in agar cum dextroso et peptono et extracto levidinis (GPY) post unum mensem candida ad crenea, obscura, laevis, margine integri. In agar cum extracto malti (YM) post 3–5 dies ad 25 °C cellulae sphaeroideae ad ovoideae ($2\text{--}3 \times 2.5\text{--}5\text{ }\mu\text{m}$), singulae, binae vel aggregatae, multilateraliter gemmantes. Post hebdomades tres in agar cum extractis levidinis et malti (YM) vel in agar cum extracto Solani tuberosi et dextroso (PD) ad 20–25 °C pseudohyphae vel hyphae verae desunt. Status teleomorphicus post cultivationem tres dies in agar cum dextroso et peptono et extracto levidinis dein post cultivationem tres dies in agar cum extracto malti et peptono (MYP) dein post cultivationem hebdomadam unum in agar cum dextroso et peptono et extracto levidinis observatus, homothallicus. Asci conjugatione cellularum gemmarumque oriuntur. Asci continentens 2–4 pileiformes sporas. Glucosum fermentatur. D-Glucosum, L-sorboseum, D-xylosum, L-rhamnosum, cellobiosum, salicinum, glycerolum, ethanolum, D-glucitolum, D-mannitolum, acidum succinicum et acidum DL-lacticum assimilantur at non D-galactosum, D-glucosaminum, L-arabinosum, D-arabinosum, sucrosus, D-maltosus, a, a-trehalosus, lactosus, raffinosis, melezitosis, inulinum, amyllum solubile, erythritolum, ribitolum, myo-inositolum nec acidum citricum. Assimilatio kalii nitrati, natrii nitrosi, L-lysini et ethylamini. Materia amyloidea non formatur. Ureum non finditur. Vitamina externa ad crescendum necessaria. Temperatura maxima crescentiae: 28 °C, 30 °C (lente).

Etymology. The specific epithet '*vustinii*' is in honour of the Russian zoologist Dr Michael M. Vustin for his contributions to the studies of *Williopsis* species and his suggestions concerning the observation of sexual stages of this novel species.

On Glucose Peptone Yeast extract Agar (GPYA), after 1 mo at 25 °C, the streak culture is white to cream, dull and smooth. The margin is entire. After growth on Yeast extract Malt extract malt (YM) agar for 3–5 d at 25 °C, cells are spheroidal to short ovoidal ($2\text{--}3 \times 2.5\text{--}5\text{ }\mu\text{m}$), occurring singly, in pairs or in small clusters, and proliferating by multilateral budding. Pseudohyphae and true hyphae are not observed after 3 wk in plate culture, neither on YM nor on potato-dextrose (PDA, Difco) agar at 20–25 °C. The sexual stage was obtained for all studied strains after consequent incubation of the culture on GPYA, Malt extract Yeast extract Peptone (MYP) agar and GPYA (3–7 d on each medium). Ascus formation preceded by conjugation between a parent cell and a bud. Asci contain 2–4 hat-shaped or saturn-shaped ascospores.

Glucose is fermented. Assimilation of carbon compounds: D-glucose, L-sorbose, D-xylose, L-rhamnose, cellobiose, glycerol, ethanol, D-glucitol, D-mannitol, DL-lactate and succinate. No growth occurs on D-galactose, D-glucosamine,

L-arabinose, D-arabinose, sucrose, D-maltose, a,a-trehalose, lactose, raffinose, melezitose, inulin, soluble starch, erythritol, ribitol, myo-inositol, D-glucuronate, citrate, D-glucarate and L-Tartaric acid. Assimilation of nitrogen compounds: potassium nitrate, sodium nitrite, L-lysine and ethylamine. Starch-like compounds are not produced. Urease activity is negative. Growth on vitamin-free medium is negative. Maximal growth temperature: 28 °C (normal growth), 30 °C (weak).

Typus. GERMANY, Thuringia, National Park Hainich, soil collected at the forest, 51.3558 N, 10.517 E, Apr. 2008, HEW-8-BEB; holotype, culture ex-type HEW-8-5 = CBS 11554, A. Yurkov, GenBank FN555431 (ITS-region), FN428955 (D1/D2 domain of 26S rRNA gene), FN555430 (16S rRNA gene), MycoBank MB515234.

Notes — Yeasts with saturn-shaped ascospores, formerly accommodated in the genus *Williopsis*, have been observed in soils and rhizospheres in different regions of the world¹. Several attempts have been made to reclassify this genus due to certain heterogeneity in their physiology¹, life cycle², distribution in different soil types³ and ribosomal gene sequence analyses^{1,4}. Recently, on the basis of multigene phylogenetic analysis, the genera *Pichia* and *Williopsis* were re-classified⁴. *Williopsis californica*, *W. pratensis* and several members of the genus *Pichia* were transferred to the novel genus *Barnettozyma*⁴.

During a project aimed to study yeasts in soils under different land use regimes, multiple cultures resembling morphological properties of *Williopsis californica* were isolated by plating soil suspensions on GPYA plates. Detailed physiological and phylogenetic analyses showed that the strains belong to the genus *Barnettozyma*, *Saccharomycetales* (*Ascomycota*, *Saccharomycotina*), but did not correspond to any of the hitherto recognised species.

For phylogenetic tree see MycoBank MB515234.

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Colour illustrations. Plot in the National Park Hainich and soil core sample from which the ex-type strain was isolated; differential interference contrast micrographs of *Barnettozyma vustinii*: vegetative cells and asci with saturn-shaped ascospores. Scale bars = 10 μm .

References. ¹Kurtzman CP, Fell JW (eds). 1998. The yeasts. A taxonomic study: 413–419. Elsevier, Amsterdam. ²Naumov GI, Vustin MM, Babjeva IP. 1980. Sexual divergence of yeasts of the genera *Williopsis* Zender, *Zygowilliopsis* Kudriavzev and *Hansenula* H. & P. Sydow. *Doklady Akademii Nauk SSSR* 255: 468–471. ³Vustin MM, Babjeva IP. 1981. Natural habitats of the yeast of the genera *Williopsis* Zender, *Zygowilliopsis* Kudriavzev. *Microbiology* 50: 1088–1091. ⁴Kurtzman CP, Robnett CJ, Basehoar-Powers E. 2008. Phylogenetic relationships among species of *Pichia*, *Issatchenkia* and *Williopsis* determined from multigene sequence analysis, and the proposal of *Barnettozyma* gen. nov., *Lindnera* gen. nov. and *Wickerhamomyces* gen. nov. *FEMS Yeast Research* 8: 939–954.

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Zasmidium macluricola



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***Zasmidium macluricola* R.G. Shivas, A.J. Young & U. Braun, sp. nov.**

Conidiomata sporodochialia, hypophylla, densa. Mycelium internum. Conidiophora multa, compacta, densis fasciculis tegentibus superficiem stromatum, $15\text{--}45 \times 4\text{--}6 \mu\text{m}$, 0–1-septata. Conidia sola vel catenis breviter ramosis, subcylindracea ad fusoida, recta, $15\text{--}45 \times 4\text{--}5 \mu\text{m}$, 0–3-septata.

Etymology. Named after the host plant *Maclura* (Moraceae).

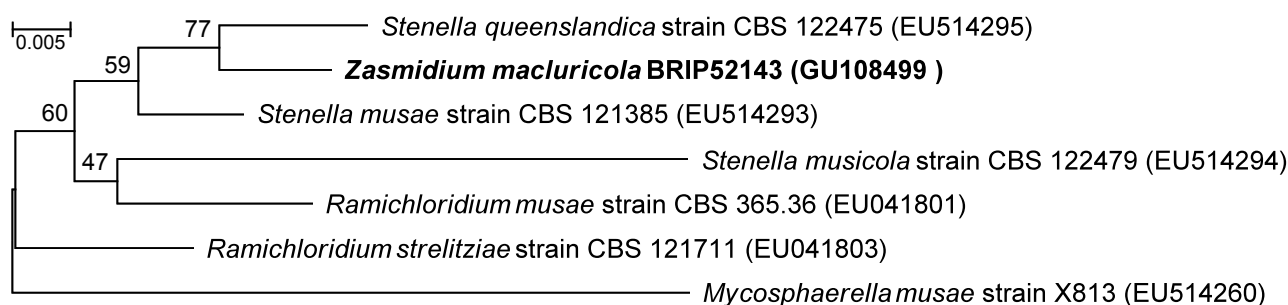
Leaf spots circular to irregular with irregular margin, up to 2 cm diam, often coalescing, covering entire leaf; upper leaf surface chlorotic, becoming pale brown and necrotic, lower leaf surface dark from abundant sporulation. *Conidiomata* sporodochial, hypophyllous, dense, punctiform, pulvinate, confluent, dark brown to black. *Mycelium* internal. *Stromata* well-developed, erumpent. *Conidiophores* numerous, compact, in dense fascicles, erect, subcylindrical to geniculate-sinuous, unbranched, $15\text{--}45 \times 4\text{--}6 \mu\text{m}$, 0–1-septate, pale reddish brown, tips paler, wall smooth; conidiophores reduced to conidiogenous cells or integrated, terminal, 10–20 μm long, proliferation sympodial, scars conspicuous, terminal and lateral, aggregated, 1.5–2 μm diam, thickened, darkened, sometimes subdenticulate, truncate. *Conidia* solitary or in short branched chains, subcylindrical to fusoid, straight, slightly curved or sinuous, $15\text{--}45 \times 4\text{--}5 \mu\text{m}$, 0–3-septate, often slightly constricted at septa, minutely verruculose, pale reddish brown, ends rounded, hila slightly thickened and darkened.

Typus. AUSTRALIA, Queensland, Conway National Park, alt. 200 m, on leaves of *Maclura cochinchinensis*, 8 Aug. 2008, S.G. Pearson, BRIP 52143, holotype; cultures ex-type BRIP 52143, GenBank GU108499; MycoBank MB515310.

Notes — The new species on *Maclura* fits well into the concept of *Stenellopsis*, which was a *Stenella*-like genus with fasciculate conidiophores and verruculose conidia, but lacking superficial mycelium. *Stenellopsis* was reduced to synonymy with *Stenella* as the formation of superficial mycelium was not

considered a reliable feature within cercosporoid hyphomycetes¹. There are about 210 species of *Stenella*, which is an aggregate of similar cercosporoid species that mostly have solitary conidiophores, arising from superficial hyphae with catenate or solitary, verruculose conidia². Placement of species in *Stenella* is problematic because it appears to have evolved more than once in *Mycosphaerella*. The new species on *Maclura* has conidiophores in dense fascicles which is not typical for *Stenella* in its traditional circumscription. *Cercospora prosopidis*, which is morphologically similar to the *Maclura* hyphomycete in its well-developed stromata, fasciculate conidiophores, but lacking superficial mycelium was transferred to *Stenella*.

ITS sequence analysis indicates that the new species is closely related to *S. queenslandica*, isolated from *Musa banksii* in north Queensland, Australia, and is positioned within a clade containing *Z. cellare* (the type species of *Zasmidium*) as well as several species of *Stenella* and *Ramichloridium* within the *Mycosphaerellaceae*³. *Zasmidium* is the oldest name for *Stenella*-like hyphomycetes in the *Mycosphaerellaceae* as well as being morphologically and phylogenetically indistinguishable from them. The type species of *Stenella*, *S. araguata*, clusters within the *Teratosphaeriaceae* and differs from *Stenella*-like *Mycosphaerella* anamorphs in having pileate conidiogenous loci (versus planate loci in *Zasmidium*). We prefer to maintain two phylogenetic genera and assign the new species on *Maclura* to *Zasmidium*, although most former *Stenella* spp. have not yet been transferred. Several anamorphic members of the *Mycosphaerellaceae* have been recorded on *Maclura*, e.g. *Mycovellosiella*, *Prathigada*, *Pseudocercospora*, *Ramularia* and *Stigmina*, which prompted us to choose a new epithet for fear of designating a future homonym as the current classification of these fungi is not stable.



Neighbour-joining tree of an ITS sequence alignment using MEGA4. The scale bar shows 0.005 changes per site, and bootstrap support values from 1 000 replicates are shown at the nodes. The species described here is printed in **bold** face. The tree was rooted to *Mycosphaerella musae* strain X813 (GenBank EU514260).

Colour illustrations. *Maclura cochinchinensis* with chlorotic leaves caused by *Z. macluricola* from the type locality; symptoms of infection by *Z. macluricola*; stromata; conidia. Scale bar = 1 cm for leaves of *M. cochinchinensis*; other scale bars = 10 μm .

References. ¹Braun U, Crous PW. 2005. Additions and corrections to names published in *Cercospora* and *Passalora*. *Mycotaxon* 92: 395–416. ²Crous PW, Braun U. 2003. *Mycosphaerella* and its anamorphs. *CBS Biodiversity Series* 1: 1–571. ³Arzanlou M, Groenewald JZ, Gams W, Braun U, Crous PW. 2007. Phylogenetic and morphotaxonomic revision of *Ramichloridium* and allied genera. *Studies in Mycology* 58: 57–93.

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Pseudocercospora avicenniae

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***Pseudocercospora avicenniae* R.G. Shivas, A.J. Young & Crous, sp. nov.**

Maculae foliorum amphigenae, angulares, venis foliorum marginatae, confluentes, interdum totam paginam foliorum operientes, superficies fit chlorotica versus pannos cinereos-brunneos ad fuscus in pagina inferiore. Mycelium internum et externum. Conidiophora dense aggregata, ramosa, orientia ex hyphis superficialibus, subcylindracea, recta ad geniculata-sinuata, $20\text{--}90 \times 3\text{--}5.5\ \mu\text{m}$. Cellulae conidiogenae terminales vel intercalares, cylindraceae, pallidae-brunneae, leves. Conidia sola, pallida-brunnea, levia, subcylindracea, apex obtusus, basis attenuata ad obconice truncata, $3\text{--}12$ septata, $30\text{--}100 \times 3\text{--}5\ \mu\text{m}$; hila et cicatrices inconspicuae.

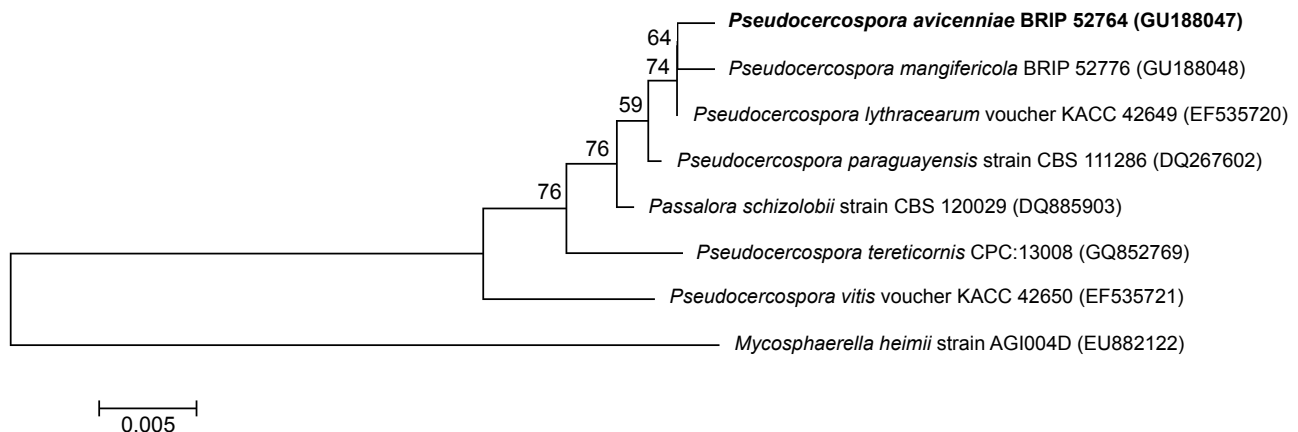
Etymology. Named after the host plant *Avicennia marina* (Acanthaceae).

Leaf spots amphigenous, angular, bordered by leaf veins, confluent, occasionally covering entire leaf surface, adaxial surface becomes chlorotic opposite greyish brown to dark brown patches on abaxial surface. **Mycelium** internal and external. **Conidiophores** densely aggregated, branched, arising from superficial hyphae, subcylindrical, straight to geniculate-sinuuous, $20\text{--}90 \times 3\text{--}5.5\ \mu\text{m}$. **Conidiogenous cells** terminal or intercalary, cylindrical, pale brown, smooth. **Conidia** solitary, pale brown, smooth, subcylindrical, apex obtuse, base attenuated to obconically truncate, $3\text{--}12$ -septate, $30\text{--}100 \times 3\text{--}5\ \mu\text{m}$; hila and scars inconspicuous.

Culture characteristics — Colonies on potato-dextrose agar (Difco) circular, up to 2 cm diam after 28 d at $25\ ^\circ\text{C}$; brownish grey to dark brown; reverse dark brown to brownish black; velvety, flat with a raised center of dense aerial mycelium, margin regular, smooth.

Typus. AUSTRALIA, Queensland, Cape Tribulation, alt. 0 m, on leaves of *Avicennia marina*, 8 Aug. 2009, R.G. & M.D.E. Shivas, P.W. & K. Crous, J. Edwards, R.C. Mann, J. Ghiano, BRIP 52764, holotype; cultures ex-type BRIP 52764, GenBank: GU188047; MycoBank: MB515468.

Notes — *Avicennia marina* (grey mangrove) is a pantropical mangrove species that is commonly found on the seaward edge of mangrove habitats across northern Australia. Molecular phylogenetic analysis supports the placement of *Avicennia* in the monogeneric family *Avicenniaceae*¹. We were unable to find records of either *Pseudocercospora* or other anamorphic *Mycosphaerellaceae* on *Avicennia*. Of interest is the close affinity of *P. avicenniae* with another newly-discovered fungus from far north Queensland, *P. mangifericola*, differing in only three bases over 500 bases of ITS1, 5.8S rRNA and ITS2 sequence. This result was validated by obtaining sequences from three separate DNA extractions from each new species.



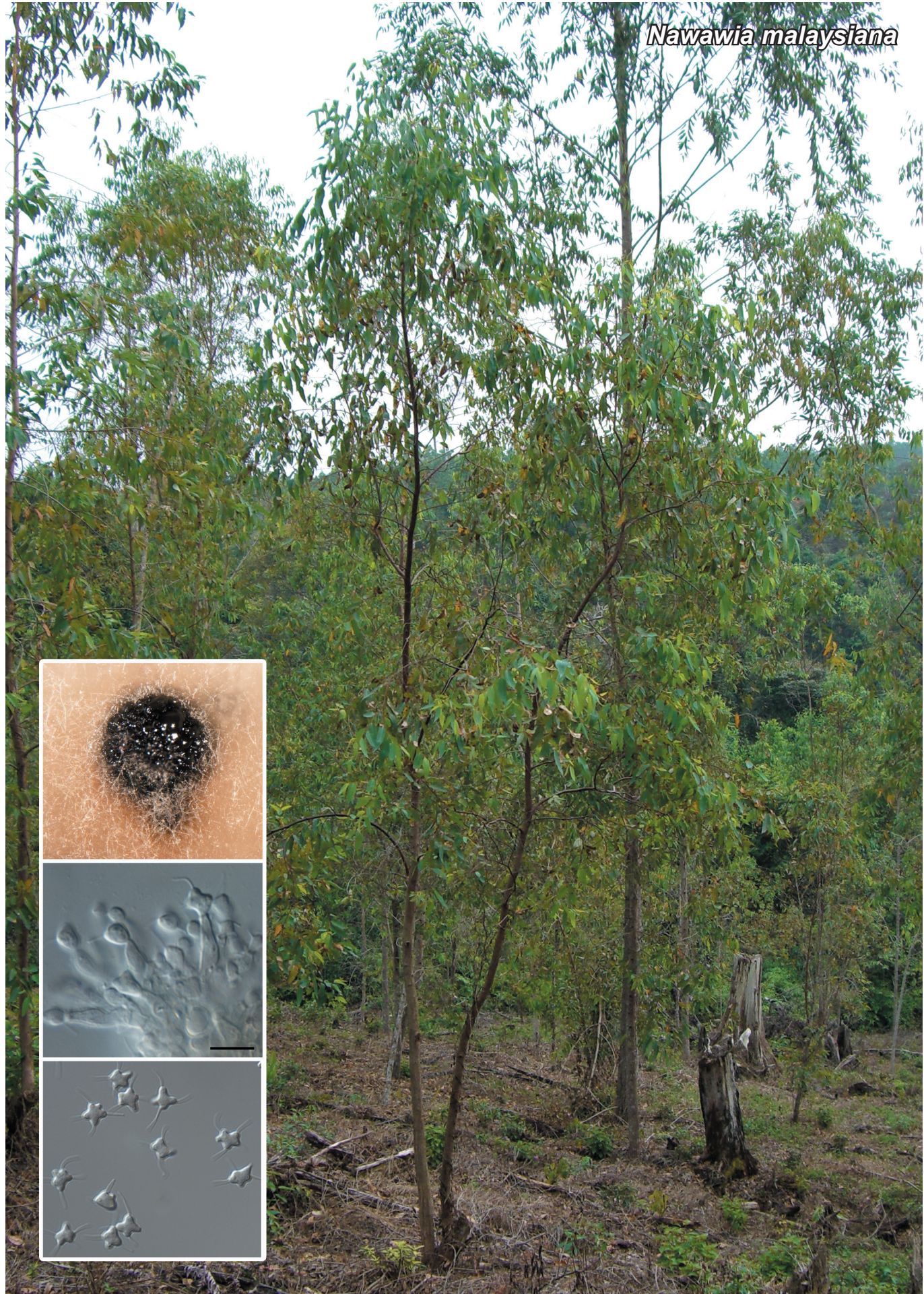
An ITS neighbour-joining tree constructed using MEGA4². The scale bar shows 0.005 changes per site, and bootstrap support values from 1 000 replicates are shown at the nodes. The species described here is printed in **bold face**. The tree was rooted to *Mycosphaerella heimii* (GenBank EU882122).

Colour illustrations. *Avicennia marina* on the foreshore at Cape Tribulation; infected leaves; stromata with conidiophores; conidia in vivo. Scale bar = 1 cm for leaves of *A. marina*; other scale bars = 10 μm .

References. ¹Schwarzbach AE, McDade LA. 2002. Phylogenetic relationships of the mangrove family Avicenniaceae based on chloroplast and nuclear ribosomal DNA sequences. *Systematic Botany* 27: 84–98. ²Tamura K, Dudley J, Nei M, Kumar S. 2007. MEGA4: Molecular Evolutionary Genetics Analysis (MEGA) v4.0. *Molecular Biology and Evolution* 24: 1596–1599.

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Nawawia malaysiana Crous & S.S. Lee, *sp. nov.*

Sporodochia ex conidiophoris dense aggregatis, subcylindraceis, 1–5-septatis composita. Cellulae conidiogenae in conidiophoris integratae, terminales vel laterales, monophialicae, 5–10 × 2.5–3.5 µm. Conidia solitaria, (4–)5(–6) µm diam. sine protuberationibus, hyalina, laevia, aseptata, quinquelobata, (4–)5(–6) protuberationibus, 1–2 µm longis, cum appendicibus gracilibus, trichoidibus, 3–9 µm longis.

Etymology. Named after the country from which it was collected, Malaysia.

Colonies sporulating on tap water agar supplemented with sterile nettle stems, on oatmeal agar (OA) and 2 % malt extract agar (MEA; Oxoid). *Conidiomata* sporodochial on all media, but at times also immersed in agar on OA, appearing semicupulate; sporodochia after 1 mo on nettle stems brown, up to 100 µm diam. *Mycelium* consisting of pale brown, smooth to somewhat roughened, septate, branched, 3–4 µm wide hyphae. *Conidiophores* pale brown, smooth, subcylindrical, densely aggregated, frequently branched, rarely loose or solitary on mycelium, 1–5-septate, 10–40 × 3–4 µm; conidiophores can also be reduced to clavate cells giving rise to 7–8 conidiogenous cells arranged like sun rays around the apex of the conidiophore cell. *Conidiogenous cells* terminal and lateral on conidiophores, monophialidic, pale brown, smooth, subcylindrical to somewhat ampulliform, 5–10 × 2.5–3.5 µm; tapering towards apex, 1–1.5 µm diam, with inconspicuous collarete (at times somewhat flaring, but mostly cylindrical), and periclinal thickening visible. *Conidia* in olivaceous, slimy masses, solitary, hyaline, smooth, round, 5-lobed, aseptate, with (4–)5(–6) projections, 1–2 µm long, that are bluntly rounded, and from the centre give rise to a thin, hair-like appendage, 3–9 µm long; conidium body (4–)5(–6) µm in height and diam; when viewed from the side conidia are fusoid to ellipsoid, 3–3.5 µm diam; the fifth conidium projection is basal, 1–2 µm long, and has a truncate base which was attached to the conidiogenous cell, 1 µm diam.

Colour illustrations. *Eucalyptus urophylla* trees growing in Malaysia; fungal colony growing on OA; conidiophores with terminal conidiogenous cells, giving rise to conidia; conidia with appendages. Scale bar = 10 µm.

Culture characteristics — Colonies on MEA at 25 °C in the dark after 2 wk: 12 mm diam, flat, spreading, with sparse aerial mycelium and feathery margins; surface smoke-grey; reverse fawn to hazel. On OA similar, with sparse aerial mycelium, reaching 15 mm diam; surface hazel.

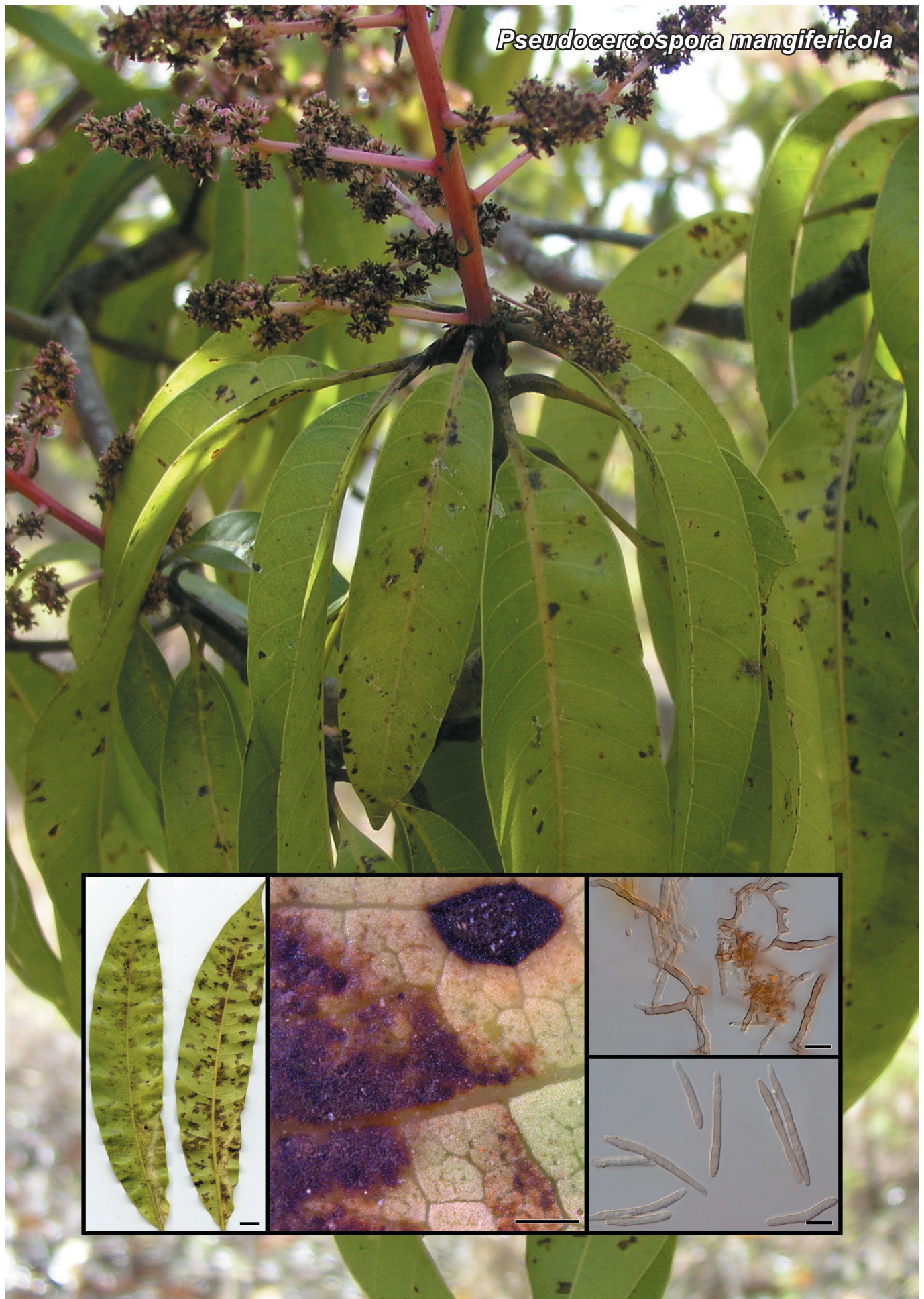
Typus. Malaysia, Sarawak, on leaves of *Eucalyptus urophylla*, 20 April 2009, S.S. Lee, CBS H-20345, holotype, cultures ex-type CPC 16757 = CBS 125544, CPC 16758; GenBank (ITS: GU229886; LSU: GU229887), MycoBank MB514707.

Notes — Four species of *Nawawia* have thus far been described, though none are known from culture. Members of the genus are recorded as being aero-aquatic¹. *Nawawia malaysiana* was isolated from the apex of a conidiomatal spore mass of a *Satchmopsis* species sporulating on a *Eucalyptus* leaf in a moist chamber. The latter provides additional support for its potential ecological role as an aero-aquatic fungus. Other species known to date form pigmented, solitary to synnematal conidiophores², and *N. malaysiana* is the first species observed to form sporodochia. However, as no other molecular data are available, this taxon is presently best accommodated in this genus.

BLASTn results of the LSU sequence were 91 % identical over approx. 910 bp with species of *Chaetosphaeria* (*Chaetosphaeriales*, *Sordariomycetes*), for example *Chaetosphaeria fuegiana* (GenBank EF063574), *Chaetosphaeria lateriphiala* (GenBank AF466071) and *Chaetosphaeria callimorpha* (GenBank AF466062).

References. ¹Descals E. 2005. Diagnostic characters of propagules in Ingoldian fungi. *Mycological Research* 109: 545–555. ²Hyde KD, Goh T-K, Steinke T. 1996. *Nawawia dendroidea*, a new synnematus hyphomycete from submerged Phragmites in South Africa. *Mycological Research* 100: 810–814.

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***Pseudocercospora mangifericola* R.G. Shivas, A.J. Young & Grice, sp. nov.**

Mycelium internum ad externum, pallidum-brunneum ad brunneum; hyphae superficiales septatae, leves, ramosae, 2.5–4.5 µm latae. Stromata absunt. Conidiophora sola vel in fasciculis laxis usque ad sex, orientia ex hyphis superficialibus, lateralibus, erecta, ampulliformia ad subcylindracea, recta vel interdum geniculata, 8–45 × 3.0–4.5 µm, raro ramosa. Cellulae conidiogenae terminales, pallidae-brunneae et pallidiores in apice. Conidia sola, pallida-brunnea, subcylindracea, 10–47 × 2.5–3.5 µm, recta vel partim curvata, apex rotundatus, basis obconice truncata ad subtruncata, 0–5-septata, interdum constricta in uno vel pluribus septis, levis.

Etymology. Derived from the name of the host plant *Mangifera* in the *Anacardiaceae*.

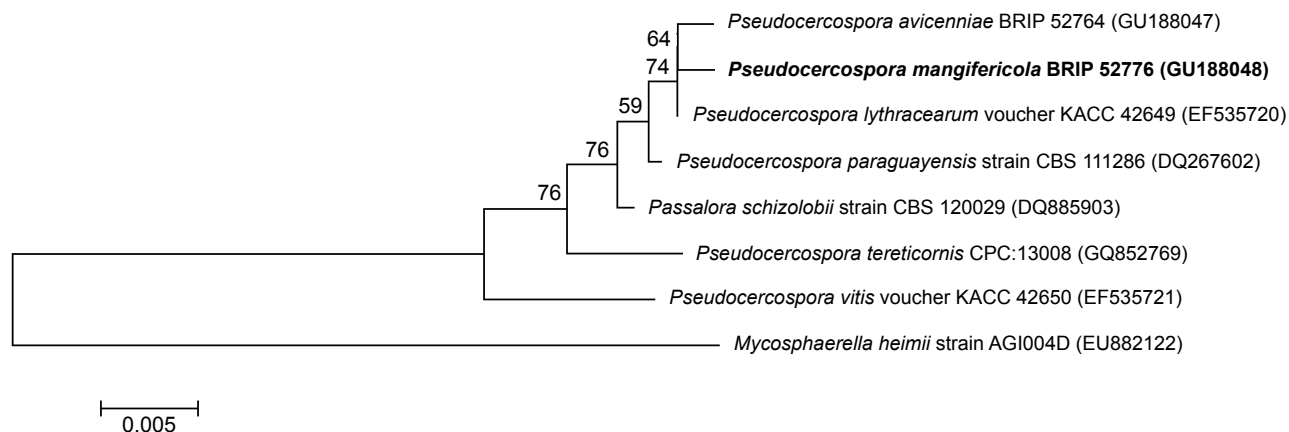
Leaf spots amphigenous; conspicuous on abaxial surface, polyangular to irregular, often vein-limited, 2–8 mm wide, margins diffuse or narrowly water-soaked, dark brown in centre becoming paler towards margin, often widespread over the entire leaf surface, lesions sometimes covered with abundant sporulation; inconspicuous on adaxial leaf surface, irregular, margins diffuse, pale to yellowish brown, always smaller than the corresponding lesion on the abaxial surface. **Mycelium** internal and external, pale to medium brown; superficial hyphae septate, smooth, branched, 2.5–4.5 µm wide. **Stromata** absent. **Conidiophores** solitary or in loose fascicles of up to 6, arising from superficial hyphae, lateral, erect, ampulliform to subcylindrical, straight or occasionally geniculate, 8–45 × 3.0–4.5 µm, rarely branched. **Conidiogenous cells** terminal, pale brown becoming paler towards the apex, proliferating percurrently or

occasionally sympodially, smooth or minutely roughened from torn annulations, rounded at apex. **Conidia** solitary, pale brown, subcylindrical, 10–47 × 2.5–3.5 µm, straight or partially curved, apex rounded, base obconically truncate to subtruncate, 0–5-septate, sometimes constricted at one or more septa, smooth; hila inconspicuous.

Culture characteristics — Colonies on malt extract agar (Difco) circular, up to 30 mm diam after 28 d at 25 °C, grey to pale olivaceous-grey, reverse olivaceous-black, radially furrowed, flat and raised in the centre, margin entire, smooth.

Typus. AUSTRALIA, Queensland, Tolga, 17° 13' S, 145° 28' E, *Mangifera indica* cv. Kensington Pride, 28 Aug. 2009, K.R.E. Grice & P. Holt, BRIP 52776b, holotype; cultures ex-type BRIP 52776b, GenBank GU188048, MycoBank MB515467.

Notes — *Pseudocercospora mangifericola* parasitised mango (*Mangifera indica*) leaves that were also infected at low levels with *Scolecotigmina mangiferae* (syn. *Cercospora mangiferae*, *Stigmata mangiferae*), which differs symptomatically by producing angular, black lesions with chlorotic haloes visible on both leaf surfaces (see middle photograph in adjacent illustrations) and is clearly distinguishable based on ITS sequence. The only other cercosporoid fungus reported on mango is *C. mangiferae-indicae*, which differs by having broader conidia measuring 3–6 µm in width¹.



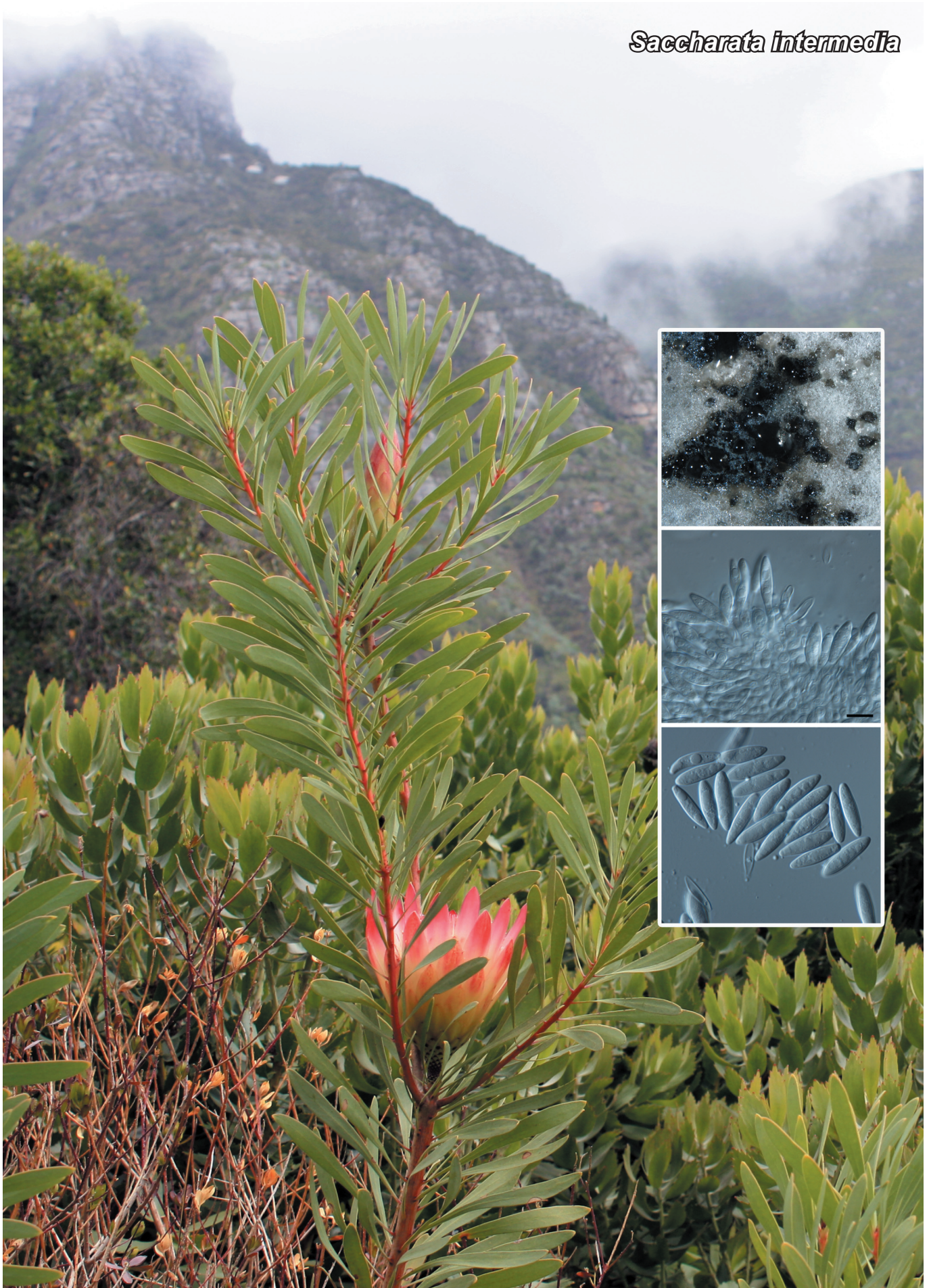
An ITS neighbour-joining tree constructed using MEGA4². The scale bar shows 0.005 changes per site, and bootstrap support values from 1 000 replicates are shown at the nodes. The species described here is printed in **bold face**. The tree was rooted to *Mycosphaerella heimii* (GenBank EU882122).

Colour illustrations. *Mangifera indica* with leaf spots caused by *Pseudocercospora mangifericola* at Tolga, Queensland; leaf (upper side left, lower side right) infected with *P. mangifericola*; leaf spots caused by *P. mangifericola* (left and middle) and *Scolecotigmina mangiferae* (top right); hyphae and conidiophores; conidia in vivo. Scale bars (from left to right) = 1 cm, 1 mm, 10 µm, 10 µm.

References. ¹Crous PW, Braun U. 2003. *Mycosphaerella* and its anamorphs. 1. Names published in *Cercospora* and *Passalora*. CBS Biodiversity Series 1: 1–157. ²Tamura K, Dudley J, Nei M, Kumar S. 2007. MEGA4: Molecular Evolutionary Genetics Analysis (MEGA) v4.0. *Molecular Biology and Evolution* 24: 1596–1599.

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Saccharata intermedia

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***Saccharata intermedia* Crous & Joanne E. Taylor, sp. nov.**

Saccharata proteae similis, sed conidiis minoribus, (17–)18–20(–22) × (3.5–)5–6 µm.

Etymology. Named after its conidial dimensions, which are intermediate in size between those of *S. proteae* and *S. kirstenboschensis*.

Conidiomata on potato-dextrose agar (PDA) pycnidial, black, up to 300 µm diam, with a single, central ostiole; wall consisting of 2–3 layers of brown *textura angularis*. **Conidiophores** subcylindrical, hyaline, smooth, frequently reduced to conidigenous cells or branched in apical part, 1–2-septate, 10–20 × 2–3.5 µm. **Conidigenous cells** terminal, subcylindrical, hyaline, 10–15 × 2–3 µm; apex with periclinal thickening, rarely with percurrent proliferation. **Paraphyses** intermingled among conidiophores, unbranched hyaline, smooth, 0–1-septate, 2–3 µm wide, extending above conidiophores. **Conidia** hyaline, smooth, fusiform to narrowly ellipsoid, apex subobtuse, base truncate with minute marginal frill, minutely guttulate, thin-walled, (17–)18–20(–22) × (3.5–)5–6 µm.

Culture characteristics — Colonies on PDA at 25 °C in the dark after 2 wk: 20 mm diam, spreading, erumpent, surface crumpled, irregular, with smooth margin and moderate aerial mycelium, lavender-grey with patches of leaden-black; leaden-grey in reverse; similar on oatmeal agar (OA) and 2 % malt extract agar (MEA; Oxoid).

Typus. SOUTH AFRICA, Western Cape Province, Kirstenbosch Botanical Gardens, on leaves of *Protea* sp., 9 Aug. 2008, J.E. Taylor, CBS H-20347, holotype, culture ex-type CPC 15557 = CBS 125546, CPC 15558, 15559; GenBank (ITS: GU229888; LSU: GU229889), MycoBank MB514708.

Notes — Three species of *Saccharata* are presently known, namely *S. proteae* (conidia 20–30 × 4.5–6 µm), and *S. capensis* (conidia 13–18 × 3.5–5.5 µm) on *Proteaceae*, and *S. kirstenboschensis* (conidia (16–)18–22(–24) × 3.5–4(–5) µm), which occurs on *Encephalartos*^{1–3}. *Saccharata intermedia*, which also occurs on *Proteaceae*, represents an intermediate species, and has conidia similar in length to those of *S. kirstenboschensis*, though somewhat wider (17–)18–20(–22) × (3.5–)5–6 µm. All species of *Saccharata* described to date occur on host plants indigenous to South Africa.

BLASTn results of the ITS sequence (Genbank GU229888) revealed that *S. intermedia* is 97 % identical to *Saccharata proteae* (Genbank FJ150708) and *Saccharata kirstenboschensis* (Genbank FJ372392) and 95 % identical to *Saccharata capensis* (Genbank EU552129). BLASTn results of the LSU sequence (Genbank GU229889) supported the association of this species with *Saccharata* with 99 % identity to both *Saccharata proteae* (Genbank EU552145) and *Saccharata capensis* (Genbank EU552129).

Colour illustrations. Collection site in Kirstenbosch Botanical Gardens, Cape Town, South Africa; fungal colony growing on PDA; conidigenous cells giving rise to conidia; fusiform conidia. Scale bar = 10 µm.

References. ¹Denman S, Crous PW, Wingfield MJ. 1999. A taxonomic reassessment of *Phyllachora proteae*, a leaf pathogen of *Proteaceae*. *Mycologia* 91: 510–516. ²Crous PW, Wood AR, Okada G, Groenewald JZ. 2008. Follicolous microfungi occurring on *Encephalartos*. *Persoonia* 21: 135–146. ³Marincowitz S, Groenewald JZ, Wingfield MJ, Crous PW. 2008. Species of *Botryosphaeriaceae* occurring on *Proteaceae*. *Persoonia* 21: 111–118.

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Xenocylindrosporium Crous & Verkley, gen. nov.

Cylindrosporio simile, sed conidiomatibus minore evolutis, conidiis curvatis, apice attenuato, 0–1-septatis et phylogenetice distinctis.

Etymology. Morphologically similar, but distinct from *Cylindrosporium*.

Conidiomata on host immersed, black, opening by irregular rupture, acervuloid, up to 300 µm diam; wall consisting of 3–4 layers of pale brown *textura angularis*. *Conidiophores* hyaline, smooth, subcylindrical, branched, septate, or reduced to ampulliform conidiogenous cells. *Conidiogenous cells* hyaline,

smooth, ampulliform to subcylindrical, terminal or lateral on septate conidiophores, monophialidic with minute periclinal thickening. *Conidia* solitary, hyaline, smooth, curved, widest in middle, tapering to acutely rounded apex and truncate base, 0–1-septate.

Type species. *Xenocylindrosporium kirstenboschense*.
Mycobank MB514709.

Xenocylindrosporium kirstenboschense Crous & Verkley, sp. nov.

Conidiomata acervulata, ad 300 µm diam. Conidiophora hyalina, laevia, subcylindrica, ramosa, 2–4-septata, 10–30 × 2–3 µm. Cellulae conidiogenae hyalinae, laeviae, ampulliformes vel subcylindricae, 5–15 × 2–3 µm. Conidia solitaria, hyalina, laevia, curvata, in medio maxime lata, apice attenuato, acute rotundato, basi truncata, 0–1-septata, (17–)22–27(–32) × (1.5–)2(–3) µm.

Etymology. Named after Kirstenbosch Botanical Gardens, South Africa, where this fungus was collected.

Leaf spots grey-brown, epiphyllous, irregular, 2–5 mm diam, coalescing. *Conidiomata* on host immersed, black, opening by irregular rupture, acervuloid, up to 300 µm diam; in culture on 2 % malt extract agar (MEA; Oxoid) similar, but pale brown; wall consisting of 3–4 layers of pale brown *textura angularis*; opening by means of irregular rupture. *Conidiophores* hyaline, smooth, subcylindrical, branched, 2–4-septate, 10–30 × 2–3 µm; or reduced to ampulliform conidiogenous cells. *Conidiogenous cells* hyaline, smooth, ampulliform to subcylindrical, terminal or lateral on septate conidiophores, 5–15 × 2–3 µm; monophialidic with minute periclinal thickening. *Conidia* solitary, hyaline, smooth, curved, widest in middle, tapering to acutely rounded apex and truncate base, 0–1-septate, (17–)22–27(–32) × (1.5–)2(–3) µm.

Culture characteristics — Colonies on MEA at 25 °C in the dark after 2 wk: 5 mm diam, spreading, erumpent, slow-growing, surface crumpled, irregular, with smooth, lobate margin and sparse aerial mycelium; surface and reverse dirty cream with patches of scarlet. On potato-dextrose agar (PDA) similar, on oatmeal agar (OA) more flattened, spreading, flesh to scarlet, with sparse aerial mycelium. In conidiomata developed on OA, microconidia were observed in addition to the typical macroconidia, produced from similar conidiomata. *Microconidia* cylindrical, hyaline, smooth, straight or curved, rounded at the top, (sub)truncate at the base, 0-septate, (5–)6–10 × 1 µm.

Colour illustrations. Leaves of *Encephalartos friderici-guilielmi* with leaf spot symptoms; leaf spot with black, immersed conidiomata; conidiophores giving rise to curved conidia; single conidiogenous cell and curved conidia. Scale bar = 10 µm.

Typus. SOUTH AFRICA, Western Cape Province, Kirstenbosch Botanical Gardens, 33° 59' 21.5" S, 18° 25' 45.4" E, on leaves of *Encephalartos friderici-guilielmi*, 13 Jan. 2009, P.W. Crous, CBS H-20346, holotype, culture ex-type CPC 16311, 16312 = CBS 125545; GenBank (ITS: GU229890; LSU: GU229891), MycoBank MB514710.

Notes — Based on its acervular conidiomata, phialides, and hyaline, curved conidia, this present collection appears to represent a species of *Cylindrosporium*, the differences being that in *Cylindrosporium* the conidiomata are shallow, and less well-developed, and the conidia are cylindrical, and aseptate. Furthermore, it also clusters apart from the type species of *Cylindrosporium*, *C. concentricum* (teleomorph *Pyrenopeziza brassicae*, *Helotiales*)¹. As *Cylindrosporium* is representative of a generic complex², a new genus, *Xenocylindrosporium*, is herewith introduced to accommodate the fungus occurring on *Encephalartos*. Although nothing is known about the ecology of *X. kirstenboschense*, it is associated with leaf spots on *E. friderici-guilielmi*, and appears to be pathogenic to this host. Although the infected plants occurred among several other *Encephalartos* species in the Kirstenbosch gardens, *X. kirstenboschense* was not observed on any other host, and may well be host specific.

BLASTn results of the ITS sequence (GenBank GU229890) revealed that the species has 85 % identity to *Phaeomoniella chlamydospora* (GenBank AB278179; incertae sedis), *Phaeomoniella capensis* (GenBank FJ372391; incertae sedis) and *Cladophialophora minutissima* (GenBank EF016377; *Chaetothyriales*); and 86 % identity to *Cyphellophora laciniata* (GenBank EU035416; *Chaetothyriales*). BLASTn results of the LSU sequence (GenBank GU229891) also supported its association with *Phaeomoniella capensis* (93 % identity; GenBank FJ372408; incertae sedis), *Amorphotheca resiniae* (92 % identity; GenBank EU030277; incertae sedis), and *Rhynchostoma proteae* (91 % identity; GenBank EU552154; incertae sedis).

References. ¹Rawlinson CJ, Sutton BC, Muthyalu G. 1978. Taxonomy and biology of *Pyrenopeziza brassicae* sp. nov. (*Cylindrosporium concentricum*), a pathogen of winter oilseed rape (*Brassica napus* ssp. *oleifera*). *Transactions of the British Mycological Society* 71: 415–439. ²Sutton BC. 1980. The Coelomycetes, Fungi imperfecti with pycnidia acervuli and stromata. Commonwealth Mycological Institute, Kew, Surrey, England.

Taxonomic novelties in this issue

Species	Gene loci sequenced
<i>Amniculicola immersa</i> Yin. Zhang, J. Fourn., Crous & K.D. Hyde, sp. nov. (p. 50)	LSU
<i>Amniculicola parva</i> Yin. Zhang, J. Fourn., Crous & K.D. Hyde, sp. nov. (p. 52)	LSU
<i>Antennariella placitae</i> Cheewangkoon & Crous, sp. nov. (p. 57)	ITS, LSU
<i>Bagadiella</i> Cheewangkoon & Crous, gen. nov. (p. 59)	ITS, LSU
<i>Bagadiella lunata</i> Cheewangkoon & Crous, sp. nov. (p. 60)	ITS, LSU
<i>Barnettozyma vustinii</i> A. Yurkov, A.M. Schäfer & Begerow, sp. nov. (p. 189)	ITS, LSU, SSU
<i>Barriopsis iraniana</i> Abdollahzadeh, Zare & A.J.L. Phillips, sp. nov. (p. 4)	ITS, EF
<i>Calonectria brachiatica</i> L. Lombard, M.J. Wingf. & Crous, sp. nov. (p. 44)	TUB, H3
<i>Calonectria brassicae</i> (Panwar & Bohra) L. Lombard, M.J. Wingf. & Crous, comb. nov. (p. 45)	TUB, H3
<i>Cladoriella paleospora</i> Cheewangkoon & Crous, sp. nov. (p. 63)	ITS, LSU
<i>Cladoriella rubrigena</i> Cheewangkoon & Crous, sp. nov. (p. 63)	ITS, LSU
<i>Clavisporea reshetovae</i> A. Yurkov, A.M. Schäfer & Begerow, sp. nov. (p. 183)	ITS, LSU, SSU
<i>Clitopilus acerbus</i> Noordel. & Co-David, nom. nov. (p. 160)	–
<i>Clitopilus albovelutinus</i> (G. Stev.) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus alutaceus</i> (Singer) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus alutaceus</i> var. <i>carpogenus</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus amarellus</i> (Cons., D. Antonini, M. Antonini & Contu) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus angustisporus</i> (Singer) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus ardosiacus</i> (E. Horak & Griesser) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus aureicystidiatus</i> (Lennox ex T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus australis</i> (Singer) Noordel. & Co-David, comb. nov. (p. 160)	–
<i>Clitopilus azalearum</i> (Murrill) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus balearicus</i> (Courtec. & Siquier) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus brunneus</i> (Contu) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus brunnescens</i> (T.J. Baroni & E. Horak) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus caelatoideus</i> (Dennis) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus carlottae</i> (Redhead & T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus carlottae</i> var. <i>vinaceus</i> (Redhead & T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus cedretorum</i> (Bidaud & Cavet) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus claudopis</i> (Singer ex T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus collybioides</i> (Singer) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus conchatus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus conicus</i> (Singer) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus crepidotoides</i> (Singer) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus crystallinus</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus cupressicola</i> (Carassai, Papa & Contu) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus cyathiformis</i> (Corner & E. Horak) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus densifolius</i> (T.J. Baroni & Ovrebo) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus dingleyae</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus eccentricus</i> (T.J. Baroni & Ovrebo) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus fibulata</i> (Pegler) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus finnmarchiae</i> (Noordel.) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus fuliginus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus fuscofarinaceus</i> (Kosonen & Noordel.) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus galerinoides</i> (Singer) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus geminus</i> (Fr.) Noordel. & Co-David, comb. nov. (p. 161)	SSU, RPB2
<i>Clitopilus geminus</i> var. <i>mauretanicus</i> (Maire) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus geminus</i> var. <i>subvermicularis</i> (Maire) Noordel. & Co-David, comb. nov. (p. 161)	–
<i>Clitopilus gibbosus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus griseolus</i> (T.J. Baroni & Halling) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus griseosporus</i> (A. Pearson) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus hawaiiensis</i> (Singer) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus heterosporus</i> (Murrill) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus himantiigenus</i> (Speg.) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus hispanicus</i> (Esteve-Rav. & G. Moreno) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus hondensis</i> (Murrill) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus horakii</i> (Pacioni & Lalli) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus hygrophoroides</i> (T.J. Baroni & Halling) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus ilicicola</i> (Lonati) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus incarnatus</i> (T.J. Baroni & Halling) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus iti</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus lactariiformis</i> (Singer) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus laetus</i> (Singer) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus lateralipes</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus lateritius</i> (T.J. Baroni & G.M. Gates) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus luteocinnamomeus</i> (T.J. Baroni & Ovrebo) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus lutetianus</i> (E.-J. Gilbert) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus mairei</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 162)	–

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Species	Gene loci sequenced
<i>Clitopilus maleolens</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus marasmioides</i> (Singer) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus melleus</i> (T.J. Baroni & Ovrebo) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus melleopallens</i> (P.D. Orton) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus mordax</i> (G.F. Atk.) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus multilamellatus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus muritai</i> (G. Stev.) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus mustellinus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 162)	–
<i>Clitopilus mycenoides</i> (Singer) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus naucoria</i> (Singer) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus nitellinus</i> (Fr.) Noordel. & Co-David, comb. nov. (p. 163)	LSU, SSU, RPB2
<i>Clitopilus nitellinoides</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus nuciolens</i> (Murrill) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus obscurus</i> (Pílát) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus obtusatus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus ochraceopallidus</i> (Ballero & Contu) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pallens</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pallidogriseus</i> (T.J. Baroni & G.M. Gates) Noordel. & Co-David, comb. nov. (p. 163)	LSU, SSU, RPB2
<i>Clitopilus parilis</i> var. <i>wagramensis</i> (Hauskn. & Noordel.) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus paurii</i> (T.J. Baroni, Moncalvo, R.P. Bhatt & S.L. Stephenson) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus peculiaris</i> (Contu & Bon) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pegleri</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus perplexus</i> (T.J. Baroni & Watling) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus perstriatus</i> (Corner & E. Horak) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus piperatus</i> (G. Stev.) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pleurogenus</i> (Pegler) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus porcelanicus</i> (Dennis) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus praesidentialis</i> (Cons., Contu, M. Roy, Selosse & Vizzini) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus priscuus</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pseudonitellinus</i> (Dennis) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus pseudopiperitus</i> (T.J. Baroni & G.M. Gates) Noordel. & Co-David, comb. nov. (p. 163)	LSU, SSU, RPB2
<i>Clitopilus pulchrispermus</i> (T.J. Baroni & Halling) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus radicans</i> (Cleland) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus reticulatus</i> (Cleland) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus rhizogenus</i> (T.J. Baroni & E. Horak) Noordel. & Co-David, comb. nov. (p. 163)	–
<i>Clitopilus russularia</i> (Singer) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus semiarboricola</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus stanglianus</i> (Bresinsky & Pfaff) Noordel. & Co-David, comb. nov. (p. 164)	LSU, RPB2
<i>Clitopilus stipitatus</i> (A.H. Sm. & Hesler) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus subcaespitosus</i> (Esteve-Rav.) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus tasmanicus</i> (T.J. Baroni & G.M. Gates) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus tergipes</i> (Corner & E. Horak) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus testaceus</i> (Dennis) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus tillii</i> (Krisai & Noordel.) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus umbrosus</i> (T.J. Baroni & Halling) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus variisporus</i> (Voto) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus verrucosus</i> (Thiers) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Clitopilus villosus</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Cyphellophora eucalypti</i> Cheewangkoon & Crous, sp. nov. (p. 63)	ITS, LSU
<i>Dacrymyces ancyleus</i> Shirouzu & Tokum., sp. nov. (p. 22)	LSU
<i>Dacrymyces aureosporus</i> Shirouzu & Tokum., sp. nov. (p. 22)	LSU
<i>Dacrymyces pinacearum</i> Shirouzu & Tokum., sp. nov. (p. 26)	LSU
<i>Dacrymyces subarcticus</i> Shirouzu & Tokum., sp. nov. (p. 28)	LSU
<i>Dacryopinax sphenocarpa</i> Shirouzu & Tokum., sp. nov. (p. 29)	LSU
<i>Elinoë eucalypticola</i> Cheewangkoon & Crous, sp. nov. (p. 64)	ITS, LSU
<i>Entoloma abbreviatipes</i> (Largent) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma aciculocystis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma acuferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma acutoumbonatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma acutum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma afrum</i> (Pegler) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma albivellum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma alboroseum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma alboroseum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma ambiguum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma andersonii</i> (Mazzer) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma angustisporum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma anisothrix</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–
<i>Entoloma applanatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 164)	–

Taxonomic novelties in this issue (cont.)

Species	Gene loci sequenced
<i>Entoloma approximatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma arcuatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma argenteolanatum</i> (T.J. Baroni, Perd.-Sánchez & S.A. Cantrell) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma asterospermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma asterosporum</i> (Coker & Couch) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma atrifucatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma atrovelutinum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma atrovelutinum</i> var. <i>leiopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma atroviolaceum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma atypicum</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma avellanicolor</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma azureum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma badissimum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma bicoloripes</i> (Largent & Thiers) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma bisporiferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma bituminosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma brunneolamellatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma brunneoloroseum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma caeruleonigrum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma caesiolumbatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma caesiomurinum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma caesiopileum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma callidermoides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma callidermum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma callithrix</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma callithrix</i> var. <i>lasiopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 165)	–
<i>Entoloma calongei</i> (E. Horak & G. Moreno) Noordel. & Co-David, comb. nov. (p. 166)	LSU, SSU
<i>Entoloma campanulatum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma candicans</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma capitatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma capnoides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma celatum</i> (Mazzer) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma chilense</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma chloroconus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma chloroides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma chlorospilum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma ciliferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cinereovirens</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma clavipilum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma coactum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma coeleste</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma coelopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma concavum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma coprinoides</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cremeoluteum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma crenulatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cristalliferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cupressum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma curtissimum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cyananthes</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cyanocalix</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cyanoides</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 166)	–
<i>Entoloma cyathus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma cylindrocapitatum</i> (T.J. Baroni & Ovrebo) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma cystidioliferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma cystomarginatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma davidii</i> Noordel. & Co-David, nom. nov. (p. 167)	–
<i>Entoloma debile</i> (Corner & E. Horak) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma deconicoides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma decurrentius</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma deformisporum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma denticulatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dichrooides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dicubospermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dimorphocystis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma diversum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dochmiopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dochmiopus</i> var. <i>obsoletum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma domingense</i> (T.J. Baroni) Noordel. & Co-David, comb. nov. (p. 167)	–

Taxonomic novelties in this issue (cont.)

Species	Gene loci sequenced
<i>Entoloma dryophiloides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dubium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma dulcisaporum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma ealaense</i> (Beeli) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma eburneum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma eburneum</i> var. <i>luteomaculatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma effugiens</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma elaeidis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
<i>Entoloma elegans</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 167)	–
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<i>Entoloma euchloroides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma eudermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
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<i>Entoloma euteles</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma exalbidum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fabaceolum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fastigiatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma ferreri</i> (T.J. Baroni, Perd.-Sánchez & S.A. Cantrell) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma ferreri</i> var. <i>pallidonigrescens</i> (T.J. Baroni & S.A. Cantrell) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fibrillosipes</i> (Murrill) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fibulatum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma flexuosipes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma foliocontusum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fructufragrans</i> (Largent & Thiers) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fulviceps</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma furfuracidiscus</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fuscatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fusciceps</i> (Kauffman) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fuscoocellatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fuscoortonii</i> (Largent) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fuscistis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma fusiferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma gabonicum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
<i>Entoloma galeoides</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 168)	–
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<i>Entoloma gigaspermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma gilvum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma glaucogilvum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma glutiniceps</i> (Hongo) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma grammatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma granulatum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma griseipes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma griseorseum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma griseovioleum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma hirtellum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma holocyaneum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma holoconiotum</i> (Largent & Thiers) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma horridum</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma humicola</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma hypochlorum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma hypoglaucum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma ianthomelas</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma incanosquamulosum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma incurvum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma infundibulare</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma inocybospermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma inodes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma insuetum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma intervenosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma invisibile</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 169)	–
<i>Entoloma ionocyanum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma ionocyanum</i> var. <i>parvipapillosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma irinum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma kansaiense</i> (Hongo) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lamellirugum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lasium</i> (Berk. & Broome) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lateripes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lateritium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–

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<i>Entoloma lecythiocystis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lecythiophorum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma leptohyphes</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma leptoniisporum</i> (Costantin & L.M. Dufour) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma leucocephalum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma leucopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lisalense</i> (Beeli) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma longissimum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lutense</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma lutulentum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma macrosporum</i> (J.W. Cribb) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma mammiferum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma margaritifera</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma mediofuscum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma megalothrix</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma membranaceum</i> (Pegler) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma microcystis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 170)	–
<i>Entoloma minutopilum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma minutostriatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma miraculosum</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma modestissimum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma modicum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma mondahense</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma nidosiforme</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma nudipileum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma nudum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma obnubile</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma obscuratum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma obscurumarginatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma ocellatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma oncocystis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma oncocystis</i> var. <i>pseudococles</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma ovatosporum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pallidius</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pallidissimum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pallidocarneau</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pallidocarneau</i> var. <i>lilliputianum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pallidosporum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pandanicola</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma pardinum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma paucifolium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma perflavifolium</i> Noordel. & Co-David, nom. nov. (p. 171)	–
<i>Entoloma perfuscum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 171)	–
<i>Entoloma phaeoxanthum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma phaeum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pigmentosipes</i> (Largent) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pilosellum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma planoconvexum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma platyspermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pluricolor</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma poliothrix</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma polyphyllum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma propinquum</i> Noordel. & Co-David, nom. nov. (p. 172)	–
<i>Entoloma pseudobulbipes</i> (Largent) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudocystidium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudodenticulatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudodochniopus</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudohirtipes</i> (Largent) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudorrhombosporum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudostictium</i> (Largent) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pseudotruncatum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pudicum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pulcherrimum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pulveripes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma punctipileum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma punctulatum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma pusillipapillatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 172)	–
<i>Entoloma quadrosporum</i> (Largent & O.K. Mill.) Noordel. & Co-David, comb. nov. (p. 172)	–

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<i>Entoloma rectangulum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma remotum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rigidipus</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rhodanthes</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rhodellum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rigens</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma roseicinnamomeum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma roseomurinum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rostratum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rotula</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rufum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma rugiferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma sabulosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma sclerobasidium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma scabulosum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma separatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma simplex</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma spermaticum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma spurium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma stylobates</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subalbidulum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subbulbosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subcapitatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subfusiferum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subglabrum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma subgracile</i> (Largent) Noordel. & Co-David, comb. nov. (p. 173)	–
<i>Entoloma sublatifolium</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subnigrellum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subrhombospermum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subrubineum</i> (Largent & B.L. Thomps.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subroseum</i> (T.J. Baroni & Lodge) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subsericeoides</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subsolitiale</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subsquamosum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subdivuense</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma subviolaceovernum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma sulcatum</i> (T.J. Baroni & Lodge) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma tenebrosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma tenuipileum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma thiersii</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma tigrinellum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma titthiophorum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma tortile</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma transitum</i> (E. Horak) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma trichomatum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma tristissimum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma truncatum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma turbidiforme</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma umbrinellum</i> (S. Imai) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma umbrosum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma variesporum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma velutipileum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 174)	–
<i>Entoloma vestipes</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma vetulum</i> (Romagn.) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma vinosulum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma violaceonigrum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma viridiflavipes</i> (Largent) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma xanthocnemis</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma xanthophaeum</i> (Romagn. & Gilles) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma xanthophyllum</i> (Largent) Noordel. & Co-David, comb. nov. (p. 175)	–
<i>Entoloma zuccherellii</i> (Noordel. & Hauskn.) Co-David & Noordel., comb. nov. (p. 175)	LSU, SSU
<i>Foliocryphia</i> Cheewangkoon & Crous, gen. nov. (p. 65)	ITS, LSU
<i>Foliocryphia eucalypti</i> Cheewangkoon & Crous, sp. nov. (p. 65)	ITS, LSU
<i>Leptoxypium madagascariense</i> Cheewangkoon & Crous, sp. nov. (p. 67)	ITS, LSU
<i>Macalpinomyces mackinlayi</i> McTaggart & R.G. Shivas, sp. nov. (p. 187)	ITS
<i>Nawawia malaysiana</i> Crous & S.S. Lee, sp. nov. (p. 195)	ITS, LSU
<i>Neofabraea eucalypti</i> Cheewangkoon & Crous, sp. nov. (p. 67)	ITS, LSU
<i>Ophiostoma denticiliatum</i> Linnakoski, Z.W. de Beer & M.J. Wingf., sp. nov. (p. 12)	ITS, TUB

Taxonomic novelties in this issue (cont.)

Species	Gene loci sequenced
<i>Parasymphodiella eucalypti</i> Cheewangkoon & Crous, sp. nov. (p. 70)	ITS, LSU
<i>Penidiella corymbia</i> Cheewangkoon & Crous, sp. nov. (p. 72)	ITS, LSU
<i>Penidiella pseudotasmaniensis</i> Crous, sp. nov. (p. 126)	ITS
<i>Penidiella tenuiramis</i> Crous & Summerell, sp. nov. (p. 127)	ITS
<i>Phaeobotryon cupressi</i> Abdollahzadeh, Zare & A.J.L. Phillips, sp. nov. (p. 6)	ITS, EF
<i>Phaeophleospora eugeniicola</i> Crous & Alfenas, sp. nov. (p. 127)	ITS
<i>Phaeothecoidea intermedia</i> Crous & Summerell, sp. nov. (p. 129)	ITS
<i>Phaeothecoidea minutispora</i> Crous & Summerell, sp. nov. (p. 129)	ITS
<i>Polyscytalum algarvense</i> Cheewangkoon & Crous, sp. nov. (p. 73)	ITS, LSU
<i>Pseudocercospora avicenniae</i> R.G. Shivas, A.J. Young & Crous, sp. nov. (p. 193)	ITS
<i>Pseudocercospora fori</i> (G.C. Hunter, Crous & M.J. Wingf.) G.C. Hunter, Crous & M.J. Wingf., comb. nov. (p. 108)	LSU
<i>Pseudocercospora mangifericola</i> R.G. Shivas, A.J. Young & Grice, sp. nov. (p. 197)	ITS
<i>Pseudocercospora paleobrunnea</i> Cheewangkoon & Crous, sp. nov. (p. 74)	ITS, LSU
<i>Pseudocercospora schizolobii</i> (M.J. Wingf. & Crous) M.J. Wingf. & Crous, comb. nov. (p. 108)	LSU
<i>Pseudocercospora tereticornis</i> Crous & Carnegie, sp. nov. (p. 129)	ITS
<i>Pseudoramichloridium brasilianum</i> (Arzanlou & Crous) Cheewangkoon & Crous, comb. nov. (p. 77)	ITS, LSU
<i>Pseudoramichloridium</i> Cheewangkoon & Crous, gen. nov. (p. 75)	ITS, LSU
<i>Pseudoramichloridium henryi</i> Cheewangkoon & Crous, sp. nov. (p. 75)	ITS, LSU
<i>Quambalaria simpsonii</i> Cheewangkoon & Crous, sp. nov. (p. 77)	ITS, LSU
<i>Rachicladosporium americanum</i> Cheewangkoon & Crous, sp. nov. (p. 77)	ITS, LSU
<i>Readeriella angustia</i> Crous & Summerell, sp. nov. (p. 132)	ITS
<i>Readeriella callista</i> (Syd.) Crous & Summerell, comb. nov. (p. 111)	LSU
<i>Readeriella dendritica</i> (Crous & Summerell) Crous & Summerell, comb. nov. (p. 111)	LSU
<i>Readeriella dimorphospora</i> (Crous & C. Mohammed) Crous, comb. nov. (p. 111)	LSU
<i>Readeriella eucalyptigena</i> Crous & Summerell, sp. nov. (p. 133)	ITS
<i>Readeriella menaiensis</i> Crous & Summerell, sp. nov. (p. 135)	ITS
<i>Readeriella minutispora</i> (Crous & Carnegie) Crous & Carnegie, comb. nov. (p. 111)	LSU
<i>Readeriella nontingens</i> (Crous & Summerell) Crous & Summerell, comb. nov.	LSU
<i>Readeriella patrickii</i> Crous & Summerell, sp. nov. (p. 111)	LSU
<i>Readeriella pseudocallista</i> Crous & Summerell, sp. nov. (p. 133)	ITS
<i>Readeriella tasmanica</i> Crous & Summerell, sp. nov. (p. 133)	ITS
<i>Saccharata intermedia</i> Crous & Joanne E. Taylor, sp. nov. (p. 199)	ITS
<i>Selenophoma australiensis</i> Cheewangkoon & Crous, sp. nov. (p. 77)	ITS, LSU
<i>Sphaceloma tectificae</i> Cheewangkoon & Crous, sp. nov. (p. 79)	ITS, LSU
<i>Strelitziana australiensis</i> Cheewangkoon & Crous, sp. nov. (p. 80)	ITS, LSU
<i>Teratosphaeria alboconidia</i> Crous & Summerell, sp. nov. (p. 136)	ITS
<i>Teratosphaeria alcornii</i> Crous, nom. nov. (p. 114)	LSU
<i>Teratosphaeria angophorae</i> (Andjic, Carnegie & P.A. Barber) Andjic, Carnegie & P.A. Barber, comb. nov. (p. 114)	LSU
<i>Teratosphaeria australiensis</i> (B. Sutton) Crous, comb. nov. (p. 114)	LSU
<i>Teratosphaeria blakelyi</i> (Crous & Summerell) Crous & Summerell, comb. nov. (p. 114)	LSU
<i>Teratosphaeria brunneotingens</i> (Crous & Summerell) Crous & Summerell, comb. nov. (p. 114)	LSU
<i>Teratosphaeria complicata</i> Crous & Summerell, sp. nov. (p. 136)	ITS
<i>Teratosphaeria considerianae</i> (Crous & Summerell) Crous & Summerell, comb. nov. (p. 115)	LSU
<i>Teratosphaeria corymbiae</i> (Carnegie, Andjic & P.A. Barber) Carnegie, Andjic & P.A. Barber, comb. nov. (p. 115)	LSU
<i>Teratosphaeria destructans</i> (M.J. Wingf. & Crous) M.J. Wingf. & Crous, comb. nov. (p. 115)	LSU
<i>Teratosphaeria eucalypti</i> (Cooke & Massee) Crous, comb. nov. (p. 115)	LSU
<i>Teratosphaeria lillianiae</i> (J. Walker, B. Sutton & Pascoe) Crous & Andjic, comb. nov. (p. 115)	LSU
<i>Teratosphaeria macowanii</i> (Sacc.) Crous, comb. nov. (p. 115)	LSU
<i>Teratosphaeria majorizuluensis</i> Crous & Summerell, sp. nov. (p. 137)	ITS
<i>Teratosphaeria miniata</i> Crous & Summerell, sp. nov. (p. 139)	ITS
<i>Teratosphaeria multiseptata</i> (Carnegie) Carnegie, comb. nov. (p. 115)	LSU
<i>Teratosphaeria obscuris</i> (P.A. Barber & T.I. Burgess) P.A. Barber & T.I. Burgess, comb. nov. (p. 115)	LSU
<i>Teratosphaeria profusa</i> Crous & Carnegie, sp. nov. (p. 139)	ITS
<i>Teratosphaeria stellenboschiana</i> (Crous) Crous, comb. nov. (p. 115)	LSU
<i>Teratosphaeria syncarpiae</i> (Carnegie & M.J. Wingf.) Carnegie & M.J. Wingf., comb. nov. (p. 115)	LSU
<i>Teratosphaeria viscidus</i> (Andjic, P.A. Barber & T.I. Burgess) Andjic, P.A. Barber & T.I. Burgess, comb. nov. (p. 115)	LSU
<i>Teratosphaeria wingfieldii</i> (Crous) Crous, comb. nov. (p. 115)	LSU
<i>Teratosphaeria xenocryptica</i> Crous & M.J. Wingf., sp. nov. (p. 139)	ITS
<i>Tilletia challinorae</i> McTaggart & R.G. Shivas, sp. nov. (p. 185)	ITS
<i>Ulocladium populi</i> E.G. Simmons, G. Newcombe & A. Shipunov, sp. nov. (p. 181)	ITS
<i>Xenocylindrosporium</i> Crous & Verkley, gen. nov. (p. 201)	ITS, LSU
<i>Xenocylindrosporium kirstenboschense</i> Crous & Verkley, sp. nov. (p. 201)	ITS, LSU
<i>Zasmidium aerohyalinosporum</i> Crous & Summerell, sp. nov. (p. 144)	ITS
<i>Zasmidium anthuriicola</i> (U. Braun & C.F. Hill) Crous & U. Braun, comb. nov. (p. 104)	LSU
<i>Zasmidium citri</i> (Whiteside) Crous, comb. nov. (p. 105)	LSU
<i>Zasmidium lonicericola</i> (Y.H. He & Z.Y. Zhang) Crous & U. Braun, comb. nov. (p. 140)	ITS
<i>Zasmidium macluricola</i> R.G. Shivas, A.J. Young & U. Braun, sp. nov. (p. 191)	ITS
<i>Zasmidium nabiaceense</i> Crous & Carnegie, sp. nov. (p. 142)	ITS
<i>Zasmidium nocoxi</i> Crous, sp. nov. (p. 141)	ITS
<i>Zeloasperisporium eucalyptorum</i> Cheewangkoon & Crous, sp. nov. (p. 81)	ITS, LSU