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Entomophthorales Fungi Parasitizing Sucking Insects in Egypt

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Authors' contributions

This work was carried out in collaboration among all authors. All authors read and approved the final manuscript.

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Review Article

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ABSTRACT

Entomophthorales are insect pathogenic fungi significant biological control potentials due to their high insect toxicity. This review focuses on the survey and morphological descriptions of entomophthoralean species attacking insect pests in Egypt. Until now 10 species of Entomophthorales fungi, belonging to three families (Entomophthoraceae, Neozygitaceae and Ancylistaceae) have been reported to suck insects as their hosts. These fungi are widely distributed in various climatic conditions in several Governorates, representing Lower and Upper Egypt. The fungi are the only pathogens that regularly and effectively control sucking insect populations in the natural ecosystems and agroecosystems. The present review emphasizes more studies and isolations of Entomophthorales species by using modern identification techniques so that their epidemiology and control potentials can be predicated on their role against insect pests under variable climatic conditions in Egypt. The possible relationship between population densities of sucking insect pests and Entomophthorales can be further studied to explore their effective applications under variable climatic conditions in the country.

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Keywords: Biological control; entomophthorales fungi; sucking insects; aphids.

1. INTRODUCTION

Sucking insect species considered one of major economic importance for several crops, causing plant stress, distortion, shoot stunting, and gall formation, or transmitting plant virus pathogens(Nadeem et al., 2023 , Sewify, and Ezz, , [1]. Entomophthorales are insect pathogenic fungi, which have significant biocontrol potentials against insect pests [2-4]. The Entomophthorales, belonging to the Subdivision Zygomycotina and Class Zygomycetes, include six families i.e., Entomophthoraceae, Neozygitaceae, Completoriaceae, Ancylistaceae, Meristacraceae and Basidiobolaceae [5]. The most important ones are Entomophthoraceae and Neozygitaceae with 200-300 and 15 species respectively [6].

The Family Ancylistaceae containing a genus *Conidiobolus,* includes 12 genera, the Family Neozygitaceae has two and the family Meristacnaceae has only one genus. So far, 223 species have been described, of which 195 belongs to the Entomophthoraceae family, 17 species to Neozygitaceae family, ten species to Ancylistaceae family, 35 species to the genus *Tarichium* [7-12]. One hundred and seventy-six Entomophthorales species are pathogenic to insects, nine species to spiders and seven of them parasitize mites [10]. Most of the above species (34.4%) attack members of the order Diptera, followed by the members (24.3%) of Homoptera (white flies, aphids, and scale insects).

Species of Entomophthorales were also identified from other insect orders, e.g., Lepidoptera, Coleoptera, Heteroptera, Hymenoptera, Orthoptera, and Dermaptera [9-11]. Some species were also identified from Collembola (Apterygota) [13,14].

Entomophthorales cause localized and widespread epizootics in hemipterous and homopterous insects, particularly aphids and leafhoppers, besides other insects such as grasshoppers, flies, beetles, and caterpillars Humber, [5] Humber, [14], Barta & Cagáň, [7]. Entomophthorales have shown significant biological control potential against agricultural insect pests due to their high efficiency and efficacy [15]. Entomophthorales fungi possess beneficial traits of insect biological control, such as easy mass production, short cycle infection, high rates of spores (germs) germination, and instantaneous lethal effects on insect populations and mortality occurring overnight [16]. Entomophthorales fungi are promising due to their ability to mass produce and act as insect biocides [17,18]. They lead to sustainable agricultural practices, protecting ecosystems and the environment [19].

Distinguished by their prey specificity, they do not pose a threat to non-target organisms [16] The epizootics, caused by entomophthoralean species, are most witnessed in flies, aphids, grasshoppers, caterpillars, mosquitoes, cicadas, and mites [20]. The rapid separation of the active conidia spores is an important characteristic of this fungal group [21]. Not much attention was given to Entomophthorales fungi and the information on their biological control potentials is sparse in Egypt, although few of the earlier studies have shown their impact on insect pest populations and their effectiveness as insect biopesticidal agent [22-25].

In the present review, various aspects of Entomophthorales fungi are discussed. Aspects, such as their morphology, survey, biological control potentials, and their effectiveness against insect pests have been discussed. The present review will help entomologists, horticulturists, agriculturists, and other professionals directly or indirectly associated with agriculture in Egypt.

2. SURVEY AND DISTRIBUTION OF ENTOMOPHTHORALES FUNGI IN EGYPT

The available literature reveals 10 species of Entomophthorales fungi present in Egypt (Table 1). Those reported were *Neozygites fresenii* (Nowakowski) Batko (Fam.: eozygitaceae), *Pandora neoaphidis* (Remaud & Hennebert) Humber, *Pandora delphacis* (Hori) Humber, *Entomophthora planchoniana* Cornu, *Zoophthora radicans* (Brefeld) Batko, *Batkoa apiculata* (Thaxter) Humber, *Batkoa major* (Thaxter) Remaud. & S. Keller, *Conidiobolus thromboides* Drechsler, *Conidiobolus coronatus* (Costantin) Batko and *Conidiobolus Obscures* (I.M. Hall & P.H. Dunn) Remaud. & S. Keller (Family: Ancylistaceace).

Table 1. Entomophthorales fungi recorded from sucking insect species in Egypt

Family	Species	Host Insect	Host Plant	Locality Governorate	Time of Occurrence	References
Neozygitaceae	*Neozygites fresenii* (Nowakowski) Batko	*A. craccivora*	faba bean	Giza	Nov.-Dec.	Sewify, [22]
		A. craccivora	broad bean	Sharkia	Nov.	Nada, [23]
		A. craccivora	Broad bean	Dakahlia	Dec.	Ibrahim et al., [26]
		S. graminum	wheat plants	Assiut	Feb.-March	Moubasher et al., [24]
		R. padi	wheat plants	Assiut		
		B. brassicae	canola plants	Assiut		
		R. padi	wheat plants	Assiut	Feb.- March	El-Maraghy et al., [27]
		R. padi	wheat plants	Assiut	Jan. –Feb.	Mohamed et al., [25]
		R. maidis	wheat plants	Assiut	Jan. –Feb.	
		S. graminum	wheat plants	Assiut	Jan. –Feb.	
Entomophthoraceae	*Pandora neoaphidis* (Remaud. & Hennebert)	*R. padi*	graminaceous weeds	Giza	March	Sewify and Ezz, [22]
		R. padi	-	Assiut	-	Abdel-mallek et al., [28]
		R. maidis	-			
		S. graminum	-			
		S. germanium	Annual sow thistle Wheat barley-Wild oat	Giza	Jan.-April	Nada, [23]
		Ac. pisum	Clover- Chicory	Giza	Jan.-Feb.	
		M. persicae	Annual sowthistle	Sharkia	Dec. - Jan.	
		R. maidis	barely plants	Giza	-	El-Fatih, [29]
		M. dirhodum	barely plants	Giza	-	
		S. graminum	wheat plants	Assiut	Feb. – March	Moubasher et al., [24]
		R. padi	wheat plants			
		B. brassicae	canola plants			
		R. padi	wheat plants	Assiut	Feb. – March	El-Maraghy et al., [27]
		Sitoboin avenae	wheat plants	El-Gharbia	Dec.-April	El-Sham and El-Sheikh, [30]
		R. padi L.	wheat plants	Assiut	Jan. –Feb.	Mohamed et al., [25]
		R. maidis	wheat plants	Assiut	Jan. –Feb.	
		S. graminum	wheat plants	Assiut	Jan. –Feb.	
	Pandora delphacis (Hori) Humber	*E. decipiens*	clover	Giza	Jan.-April	Nada, [23]
	Entomophthora planchoniana Cornu	*S. graminum*	wheat, barley and wild oat	Giza	Jan.-March	Nada, [23]
		R. padi	-	Assiut	-	Abdel-Mallek et al., [28]

Family	Species	Host Insect	Host Plant	Locality Governorate	Time of Occurrence	References
		R. maidis	-			
		S. graminum	-			
		S. graminum	wheat plants	Assiut	Feb.-March	Moubasher et al., [24]
		R. padi	wheat plants			
		B. brassicae	canola plants			
		R. padi	wheat plants	Assiut	Feb.-March	El-Maraghy et al., [27]
	Zoophthora radicans (Brefeld)Batko	*R. padi*	Maize	Monufia	May	Sewify and Ezz, [1]
		R. padi	-	Assiut	-	Abdel-mallek et al., [28]
		R. maidis	-			
		S. graminum	-			
		E. decipiens	bean	Giza	Dec. -Feb.	Nada, [23]
		S. graminum	wheat plants	Assiut	Feb.- March	Moubasher et al., [24]
		R. padi	wheat plants	Assiut		
		B. brassicae	canola plants	Assiut		
		R. padi	wheat plants	Assiut	Jan.- Feb.	Mohamed et al., [25]
		corn leaf aphid, *R. maidis*	wheat plants	Assiut		
		S. graminum	wheat plants	Assiut		
	Batkoa apiculata (Thaxter) Humber	*Ac. pisum*	clover, and broad bean	Giza	Jan.- Feb.	Nada, [23]
	Batkoa major (Thaxter) Humber	*E. decipiens*	bean	Giza	Dec. - Feb.	Nada, [23]
		craccivora	Broad bean	Dakahlia	March-April	Ibrhaim et al., [26]

Table 1. Continuous

Family	*Species*	*Host Insect*	Host Plant	Locality Governorate	Time of occurrence	References
Ancylistaceae	*Conidiobolus thromboides* Drechsler	*R. padi*	graminaceous weeds	Giza	March	Sewify and Ezz, [1]
		R. padi	-	Assiut	-	Abdel-mallek et al., [28]
		R. maidis	-			
		S. graminum	-			
		N. viridula	Maize	Giza	April- June	Nada, [23]
		R. maidis	barely plants	Giza	-	El-Fatih, [29]
		M. dirhodum	barely plants	Giza	-	
	Conidiobolus coronatus (Costantin) Batko	*E. lanigera*	Apple trees	Monufia	May	Sewify and Ezz, [1]
		R. padi	-	Assiut	-	Abdel-mallek et al., [28]

Family	***Species***	***Host Insect***	**Host Plant**	**Locality Governorate**	**Time of occurrence**	**References**
		R. maidis	-			
		S. graminum	-			
		S. graminum	wheat plants	Assiut	Feb. – March	Moubasher et al., [24]
		R. padi	wheat plants	Assiut		
		B. brassicae	canola plants	Assiut		
		I. seychellarum	apple	Garbiya		Ezz, [31]
		A.aurantii	Apple	Bany Suwayf		
			Grape	Al-Fayoom		
			Guava	Ismailiya		
			Orange			
		R. padi	wheat plants	Assiut	Jan.- Feb.	Mohamed et al., [25]
		R. maidis	wheat plants			
		S. graminum	wheat plants			
	Conidiobolus Obscures (I.M. Hall & P.H. Dunn) Remaud. & S. Keller	*R. padi*	-	Assiut	-	Abdel-Mallek et al., [28]
		A. craccivora	Broad bean	Assiut	April	Ibrahim et al., [26]
			cowpea	Dakahlia	May- June	Ibrahim et al., [26]

2.1 *Neozygites fresenii* (Nowakowski) Batko

The fungus was identified based on morphological characterizes according to Humber (2005). The first record of *N. fresenii* in Egypt was on the cowpea aphids, *Aphis craccivora Koch* on fava bean plants (*Fabaceae)* in Giza Governorate (Sewify, 2000). The fungus was found in the aphid population from November - December 1989 (Table 1). *Neozygites fresenii* was also recorded on *A. craccivora* on fava bean plants in November 2004 and on broad beans in December 2008-2009 in Sharkia and Dakahlia Governorates Nada, [23,26]. Besides, they were also reported to be associated with greenbug, *Schizaphis graminum* (Rondani), bird-cherry aphid *Rhopalosiphum padi* (Linnaeus) on wheat plants and cabbage aphids, *Brevicoryne brassicae* (Linnaeus) on canola plants during February – March 2006-2007 in Assiut Governorate [24]. It was also found associated with the populations of *R. padi, R.maidis* and *S. graminum* on wheat plants from January-February, 2013-2014 in Assiut Governorate [27,25] (Table 1).

2.*2 Pandora neoaphidis* (Remaud. & Hnenebert)

The first record of *P. neoaphidis* was recorded on *R. padi* on graminaceous weeds in March in the Giza Governorate of Egypt (Sewify & Ezz, 2000). Abdel-Mallek et al. [24] and El-Maraghy et al*.,* [27] reported this fungus on *R. padi, R.maidis* and *S. graminum* in Assiut Governorate. It was also found associated with *S. germanium, Acyrthosiphon pisum* Harris, Annual sow thistle, Wheat barley-Wild oat, and Clover-Chicory during January-April, 2005 in Giza Governorate and *Myzus persicae* (Sulzer) on Annual sow thistle during December 2004- January 2005 in Sharkia Governorate [23]. The fungus was also reported from *R. maidis, Metopolophium dirhodum* (Walker), and on barely any plants in Giza Governorate [28,29,26]. recorded *P. neoaphidis* on *A. craccivora* on broad bean in December 2008 –March 2009 in Dakahlia Governorate. Moubasher et al., [24] found this fungus associated with *S. graminum*, *R. padi* on wheat plants and *B. brassicae* on canola plants during February- March 2006-2007 in Assiut Governorate. *Pandora neoaphidis* was found associated with *Sitoboin avenae* on wheat plants during December-April in El-Gharbia governorate [30], whereas on *R. padi, R. maidis* and *S. graminum* on wheat plants during January-February in Assiut Governorate [25] (Table1).

2.3 *Pandora delphacis* (Hori) Humber

Pandora delphacis was first recorded on *Empoasca decipiens* on a clover plant during January-April 2005 in Giza Governorate, Egypt [23] (Table 1).

2.4 *Entomophthora planchoniana* Cornu

The fungus, *E. planchoniana* was reported for the first time in Egypt on *S. graminum* on wheat, barley, and wild oats during January-February in Giza Governorate [23] Earlier it was found associated with *R. padi, R.maidis* and *S. graminum* in Assiut Governorate (Abdel-Mallek et al., [28], El-Maraghy et al*.,* [27] and on *A. craccivora* on broad beans during January-March 2009 in Dakahlia governorate [26]. They were also found associated with *S. graminum* and *R. padi* on wheat plants and *B. brassicae* on canola plants during February-March 2006-2007 in Assiut Governorate [24] (Table 1).

2.5 *Zoophthora radicans* (Brefeld) Batko

Zoophthora radicans was recorded first time in Egypt, parasitizing *R. padi* on maize during May in Monufia Governorate [1]. It was parasitizing *R. padi, R. maidis* and *S. graminum* in Assiut Governorate [28], *E. decipiens* on clover during December 2004 –February 2005 in Giza Governorate (Nada, 2006), *S. graminum* and *R. padi* on wheat plants, and *B. brassicae* on canola plants during February- March 2006-2007 in Assiut Governorate [24]. *Zoophthora radicans* was also reported from *Sitoboin avenae* on wheat plants during December-April in El-Gharbia Governorate [30], on *R. padi, R. maidis* and *S. graminum* on wheat plants during January -February in Assiut Governorate [25] (Table 1).

2.6 *Batkoa apiculata* (Thaxter) Humber

Batkoa apiculata was reported in Egypt from *A. pisum* on clover and broad bean during January-February 2005 [23]. (Table 1).

2.7 *Batkoa major* (Thaxter) Humber

The first record of fungus *B. major* in Egypt was on *E. decipiens* on bean plan during May-June 2003 in Giza Governorate [23]. Ibrahim et al*.,* [26] recorded *B. major* on *A. craccivora* in

Dakahlia governorate during March and April 2009 (Table 1).

2.8 *Conidiobolus thromboides* Drechsler

Conidiobolus thromboides was recorded in Egypt from *R. padi* on maize plant in Monufia Governorate Sewify and Ezz, [1] from *R. padi, R.maidis* and *S. graminum* in Assiut Governorate Abdel-Mallek et al., [28], from *Nezara viridula* (L.) on Maize plant during April- June 2003 in Giza Governorate [23] and from *R. maidis* and *M. dirhodum* on barely plants in Giza Governorate [29]. (Table 1).

2.9 *Conidiobolus coronatus* (Costantin) Batko

Conidiobolus coronatus was recorded in Egypt for the first time from *Eriosona lanigera* (Hausm) on apple trees during May at Monufia Governorate [1] It was also recorded from *R. padi, R. maidis*, *S. graminum* in Assiut Governorate [28] *Icerya seychellarum* from apple trees in Garbiya Governorate and from *Aonidiella aurantii* on apple trees in Bany Suwayf Governorate, and grape and orange trees in Al-Fayoom and Ismailiya Governorates respectively [31] (Table1).

2.10 *Conidiobolus Obscures* (I.M. Hall & P.H. Dunn) Remaud. and S. Keller

The first record of *C. obscures* in Egypt was recorded on *R. padi, R. maidis* and *S. graminum* in Assiut Governorate [28] (Table 1). Ibrahim et al*.,* [26] recorded *C. obscures* from *A. craccivora* on broad beans and cowpea at Dakahlia governorate during April-June 2009.

3. PATHOGENICITY OF ENTO-MOPHTHORALES FUNGI

There were several reports on the pathogenicity of Entomophthorales against insects. To compare the controlling methods of the Californian aphid population in cotton, two releasing methods namely: chamber inoculation of *A. gossypii* and dried *N. fresenii*-infected cotton aphid ‘‘cadavers’’ and used. Both methods successfully introduced *N. fresenii* to cotton aphids and played a significant role in *A. gossypii* control [32]. The effect of three *P. neoaphidis* isolates against *Sitobion avenae* and *Rhopalosiphum padi* was studied by Saussure et al. (2019). Shah et al., [33] reported virulence of *P. neoaphidis* varies with aphid host species, host genotype (Stacey et al., [34], Parker et al., [35]. geographic origin of the isolate [36]. It also varied even between isolates co-occurring in one aphid metapopulation [37,36]. Virulence of Two Entomophthoralean fungi, *Pandora neoaphidis* and *Entomophthora planchoniana*, to their conspecific (*S. avenae*) and Heterospecific (*R. padi*) aphid hosts was carried out by Ben Fekih, et al. [38]. For the first time, the virulence of the two species of fungi was compared; both originated from *S. avenae* cadavers. The results showed that the conspecific host, *S. avenae*, was more susceptible to *E. planchoniana* infection than the heterospecific host *R. padi*. In the case of *P. neoaphidis*, Median Lethal Time 50 for *S. avenae* was 5.0 days as compared to 5.9 days for *E. planchoniana*. The LT50 for *S. avenae* was 4.9 days, while the measured infection level in *R. padi* was always below 50 percent. Pathogenicity of *Z. radicans* against different insect pests was reported, which showed pathogenicity towards bagrada bug, *Bagrada hilaris* [39], Lepidopteran larvae Walter *et. al*., [40] and whitefly *Trialeurodes vaporariorum* [41]. The influence of the aphid-specific pathogen *Conidiobolus obscures* on the mortality and fecundity of bamboo aphids was carried out by Zhou et al., (2013). At high concentrations of conidia, high mortalities (74-91%) caused by *C. obscures* were recorded in *Takecallis taiwanus*, *Takecallis arundinariae*, *Melanaphis bambusae*, and *Metamacropodaphis bambusisucta* [41,42].

4. MORPHOLOGICAL FEATURES OF FUNGI

4.1 N. fresenii

The hyphae are spherical and the primary conidia are sub-globose, with flattened basal papilla measuring 12-20 μm x 13-15 μm (16 x14 μm) and 13-20.8 μm x 10.4 -13 μm (16.9 x 11.7 μm) for Giza and Sharkia isolates respectively. The secondary conidia, capilliconidia are almond-shaped measuring 20-30 μm x 11-14 μm (25 x 12.5 μm) and 20.8 -26 μm x 13-15.6 μm (13.4x 14.3) for Giza and Sharkia isolates respectively, which are supported by capillary conidiophores of 20-30 μm [22]. The resting spores were black to smoky-gray in color arising from conjugation between two spherical gametangia [23].

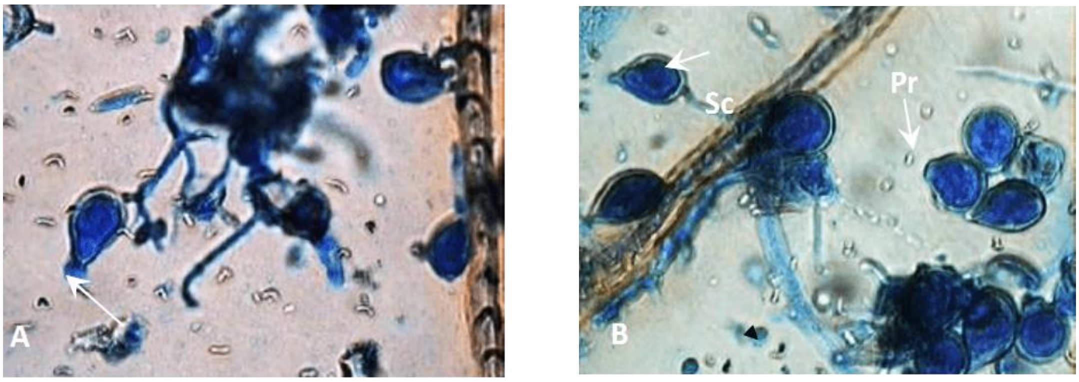

Fig. 1. Light micrographs showing *N. fresenii* infected aphid, *A. craccivora*: (A) Capilliconidia almond-shaped with a mucoid apical droplet (arrow) X1320; (B) Primary conidium with flatted basal papilla (Pr) and secondary capilliiconidum (Sc) produced on capillary conidiophore arising from primary conidium X1320[23]

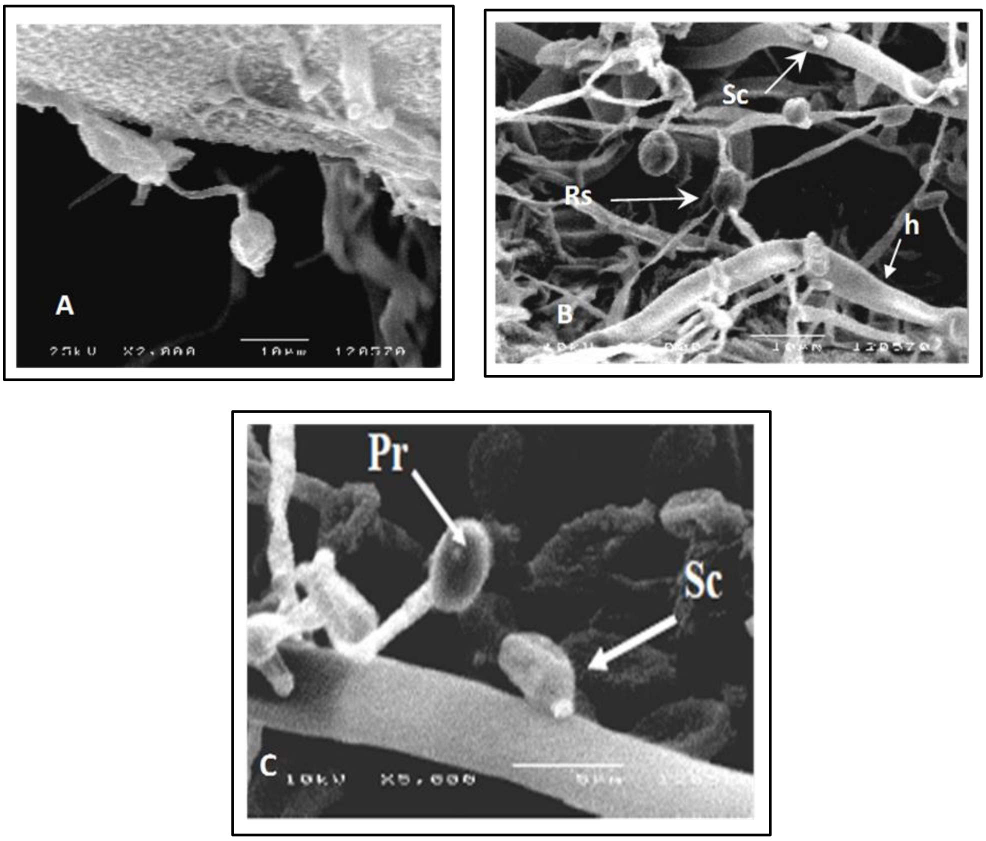

Fig. 2. Scanning electron microscopy showing *N. fresenii* infected aphid*, A. craccivora*: (A) Fungus *N. fresenii* showing secondary capilliconidium produced on capillary conidiophore arising from primary conidium X2000; (B) Fungus *N. fresenii* showing elongated hyphal body (h), capilliconidia with apical slime drop (Sc) and resting spore arising from conjugation bridge between gametangia (Rs) X2000; and (C) Fungus *N. fresenii* showing primary conidia attached to leg of *A. craccivora* X 3500

4.2 Pandora neoaphids

Conidiophores are digitately branched at the apices. Primary conidia clavate to obovoid, uninucleate with basal papilla. Secondary conidia were nearly globose, whereas Rhiziods had prominent terminal discoid holdfast. Resting spores were not observed but primary conidia of *M. persicae, S. graminum* and A. *pisum* were of varying sizes: 15.6-23.4 x 7.8–13 µm, 28.6–20.8 x 0.4–13 µm, and 20.8–26 x 10.4–13 µm respectively. The secondary conidial of S. *graminum* and A. *pisum* were 15.6–18.2 x 10.4–13 µm and 7.8–13 x 15.6–20.8 µm sizes respectively [23].

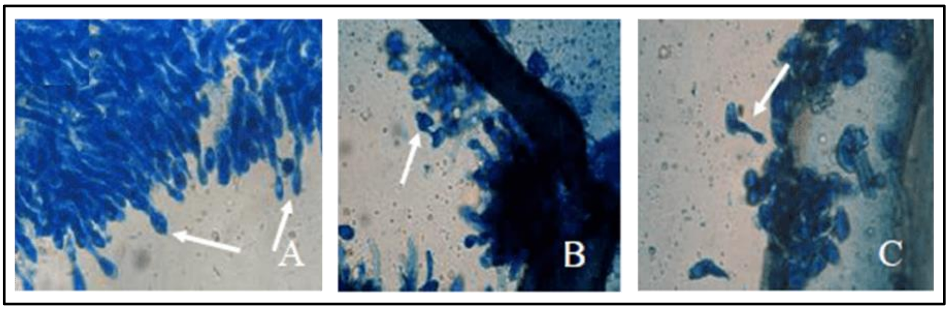

Fig. 3. Light micrographs showing *P. neoaphidis* infected aphid, *S. graminum*: (A) Conidiophores and primary conidia (arrow)of *P. neoaphidis* on *S. graminum* X600;(B-C) Primary conidia developed to secondary conidia (arrow) of fungus *P. neoaphidis* on *S. graminum* Leg X600 [23]

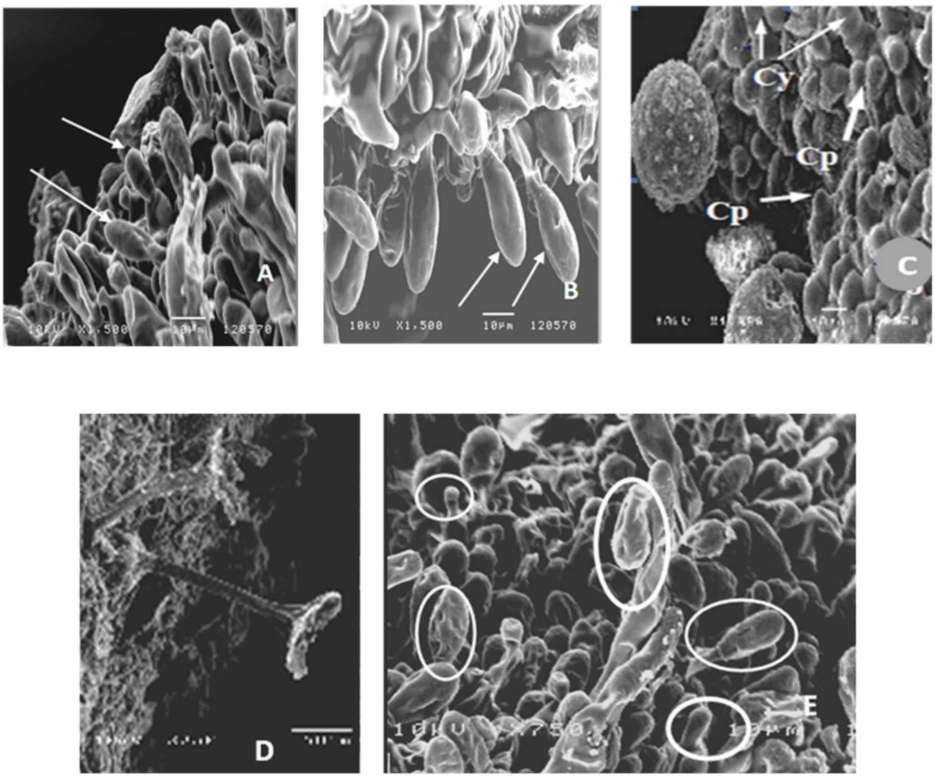

Fig. 4. Scanning electron microscopy showing *P. neoaphidis* infected aphid,*S. graminum*: (A-B) Cystidium and conidia of *P. neoaphidis* emerging from *S. graminum* head X150, 750; (C) Fungus *P. neoaphidis* showing early developing to cystidia (Cy) and conidiophores (Cp) breakthrough of the cuticle on *S. graminum* X1000, (D) Discoid terminal holdfasts of rhizoids emerging from the midventral region of infected *M. persicae* X 750; and (E)Developing process of primary conidia on *M. persicae* X1000 [23]

4.3 E*. planchoniana*

Conidiophores were simple, primary conidia had bell-shaped, flat papilla broad and pointed apexes with varying sizes (15.6-26 X 10.4 - 13µm). Secondary conidia were slightly smaller than primary conidia with variable sizes (13-18.2 X 7.8- 13µm) with rounded papillae. Rhizoids have the same diameter as conidiophores [23].

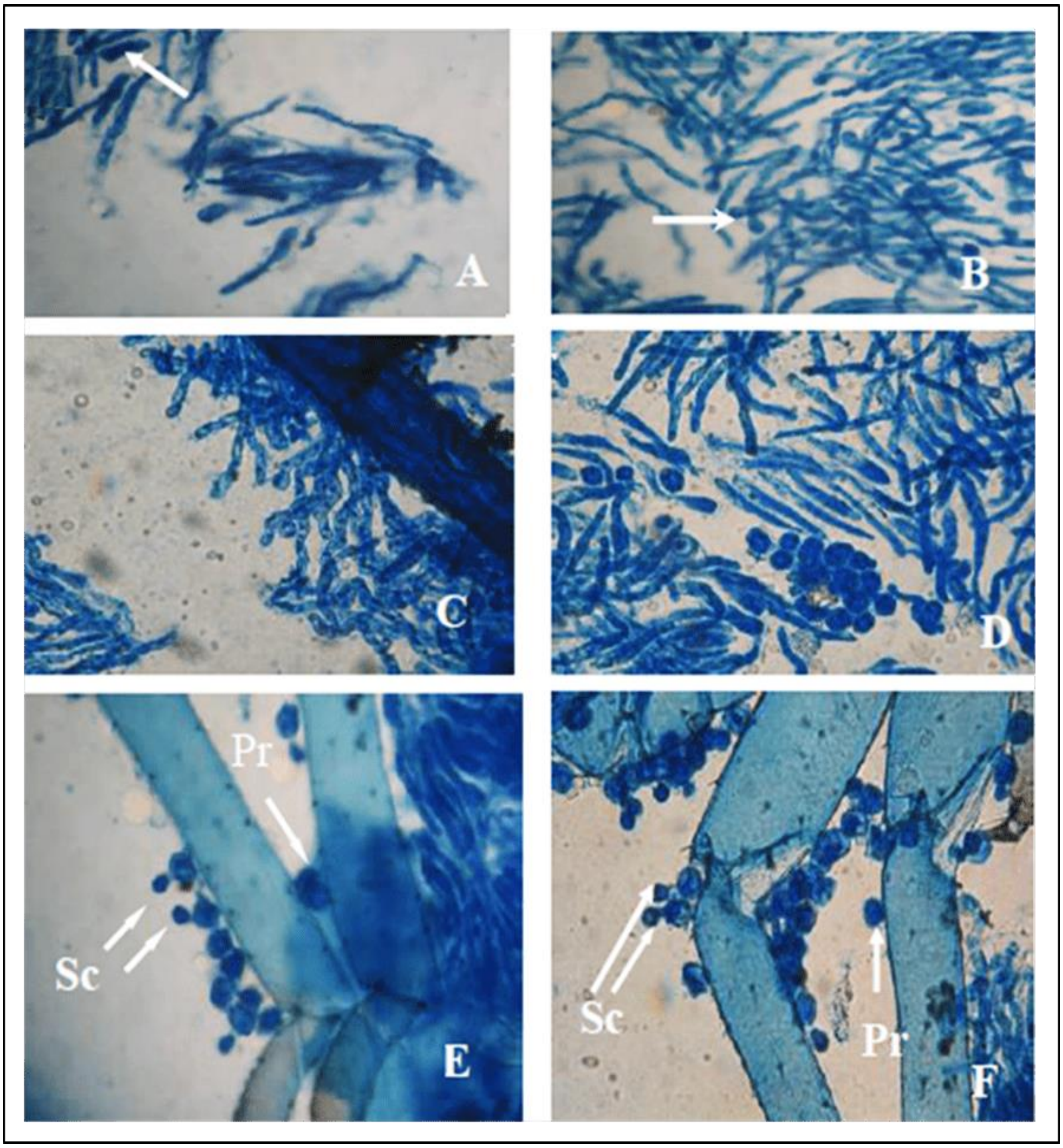

Fig. 5. Light micrographs showing *E. planchoniana* infected aphid, *S. graminum*: (A) Monohyphal rhizoids with mother cell (arrow) X600; (B) Unbranched conidiophores bearing developing conidium (arrow) X600; (C) Unbranched conidiophores of *E. planchoniana* bearing developing conidium on leg of *S. graminum* (arrow) X600; (D) Unbranched conidiophores and discharged primary conidium with apiculus X600; and (E-F) Primary conidia with apiculus (arrow), broad, nearly flat basal papilla (Pr) and secondary conidia budding from the primary conidia (Sc) X600 [23]

4.4 *B. apiculata*

Conidiophores were simple with a narrow neck between the conidium and conidiogenous cell. Primary and secondary conidia were globose, multinucleate with hemispherical papilla [23].

4.5 *Z. radicans*

Conidiophores are digitately branched with long, ovoid, and bullet-shaped conidia. The conical papilla slightly glowing or projecting when connected to the host conidia. Cystidia is

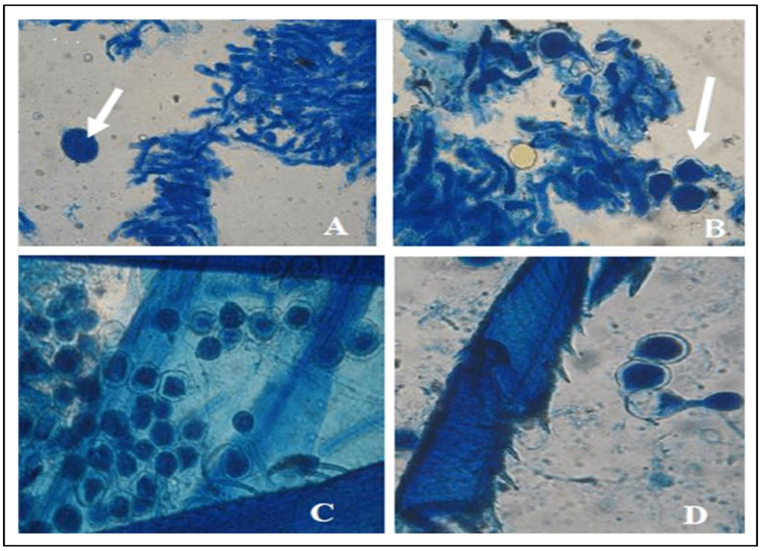

Fig. 6. Light micrograph showing *B. apiculata* infected *Ac. pisum:* (A) Fungus *B. apiculata* showing Simple conidiophores and globosely resting spores (arrow) on *Ac. pisum* X600; (B) Conidiophores and globosely primary conidia, discharged by papilla reversion (arrow) on *Ac. pisum* X600; (C) Aggregation of globosely primary conidia attached with *Ac. pisum* wing X600; and (D) Primary conidia developing to secondary conidia X600 [23]

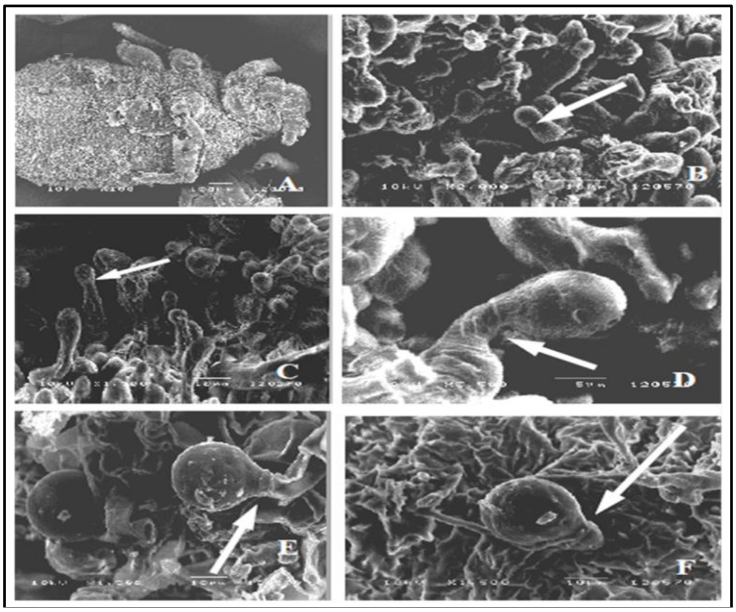

Fig. 7. Scanning electron microscopy showing *B. apiculata* infected *Ac. pisum*: (A) Conidiophores emerged through the host cuticle; (B-C) Developing conidia with narrow neck between Conidiogenous cell and conidia (arrows). (D - E) Primary conidia in the process of formation on the conidiophores (arrow). (F) Globes primary conidia discharged by papilla reversion on *Ac. pisum* (arrow) [23]

untapped towards the apex but thicker than the hyphae at the base. Rhizoids had prominent terminal discoid holdfast and primary conidia of varying sizes (13 - 20.8 X 7.8 - 10.4µm [23].

4.5 B. major

Conidiophores are simple with narrow necks between conidium and conidiogenous cells. Primary and secondary conidia were globose and multinucleate. Papilla had pointed extension and rhizoids were with terminal discoid holdfasts. Resting spores were present with rhizoids having prominent terminal discoid holdfast. The fungus was recorded in *E. decipiens*, where primary conidia varied in size (23.4- 39 X 20.8- 28.6µm). The resting spores' size was variable (18.2- 28.6 X 18.2 –28. 6µm) [23]

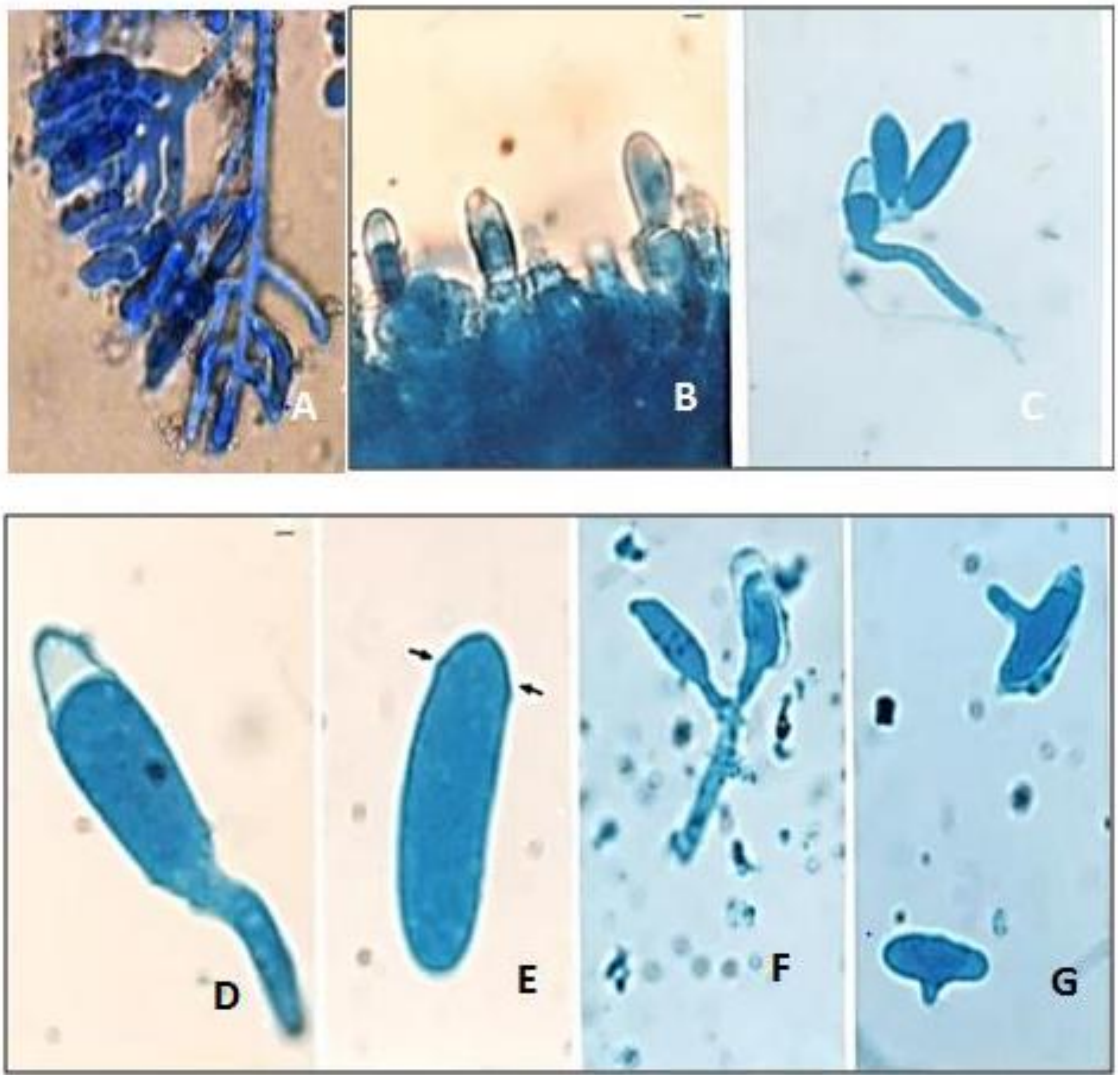

Fig. 8. Light micrographs showing *Z. radicans* infected *A. craccivora* and *E. decipiens*: (A) Branched conidiophores of *Z. radicans* on *E. decipiens* X660; (B) Emerging conidiophores and conidia from the host *A. craccivora*; (C) conidiophores and primary conidia from the host *A. craccivora*; (D-E) bullet-shaped to long ovoid conidia with a conical papilla, slight glow or projection(arrow) when the papilla is connected to the body of conidia on its host *A. craccivora,* (F) Branched conidiophores of *Z. radicans* infected *A. craccivora* (G) Germinated primary conidia of *Z. radicans* on *A. craccivora* x1320 (Nada 2006 and Sewify unpublished data)

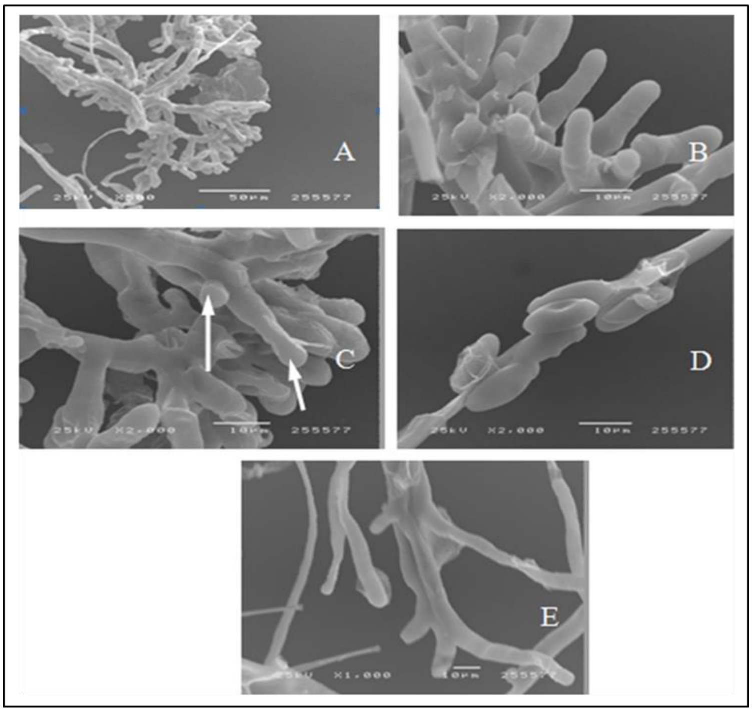

Fig. 9. Scanning electron microscopy showing *Z. radicans* infected *E. decipiens*: (A-B) Branched conidiophores of *Z.radicans* on *E. decipiens* X 500, 1000; (C) Developing process of primary conidia (arrow) of *Z. radicans* on *E. decipiens* X 2000; (D) primary conidia clavate with basal papilla rounded *E. decipiens* X 2000; and (E) Cystidium projecting from hymenium X 1000 [23]

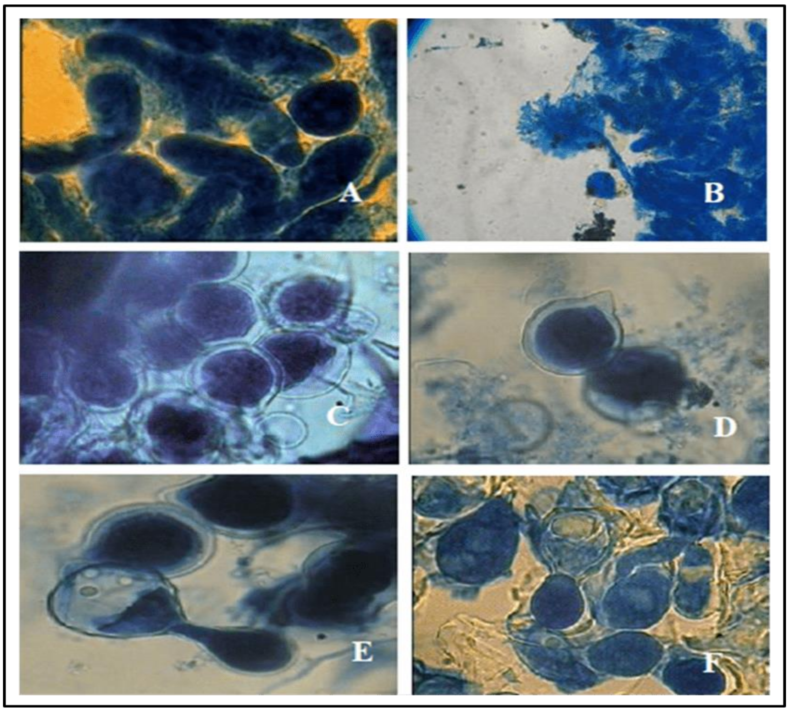

Fig. 10. Light micrographs showing *B. major* infected *E. decipiens:* (A) Simple conidiophores and globose primary conidia discharged by papilla reversion X1320; (B) Rhizoids with discoid terminal holdfasts X600; (C-D) Globosely primary conidia discharged by papilla reversion X1320; (E-F) Primary conidia developed to secondary conidia X 1320[23]

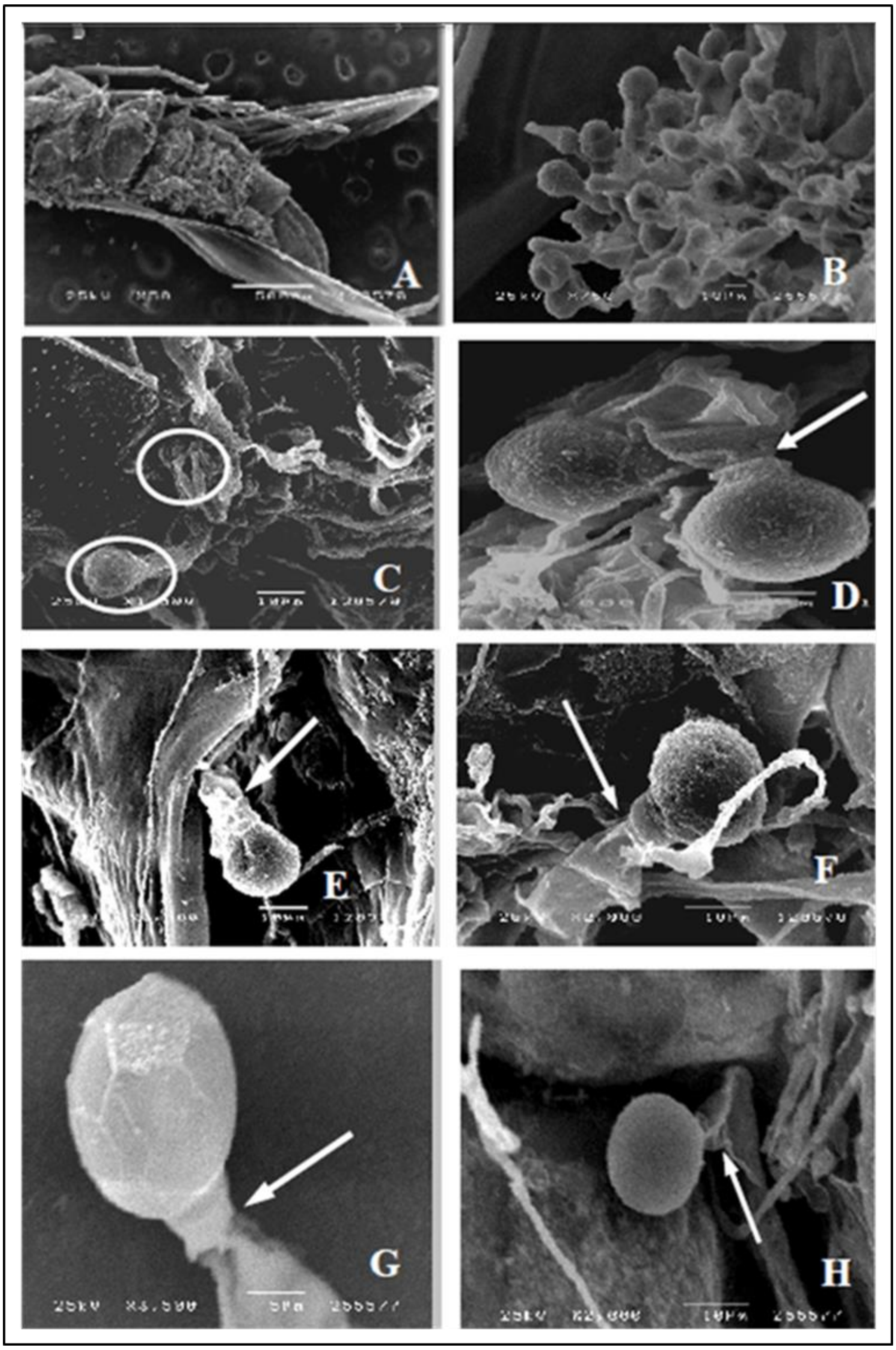

Fig. 11. Scanning electron microscopy showing B. major infected E. decipiens: (A) Overall view of mycosed E. decipiens with B. major, conidiophores emerged through the abdomen X 50; (B) Conidiophores and primary conidia of the B. major emerging through E. decipiens abdomen X 750; (C-D) Conidia of B. major in the process of formation on the conidiophores (arrows) X 1500, 1000; (E –F –G) Conidiophore of B.major ready to discharged primary conidia X 1500, 2000, 3500; and (H) Discharged primary conidia X 2000 [23]

4.6 C. thromboides

Conidiophores were simple and primary conidia were pyriform in shape with a basal papilla. Primary conidia were of variable sizes (26 - 39 X 31.2 –20.8µm), which were isolated from *N. viridula.* The resting spores measured 26- 39 X 23.4- 36.4µm [23].

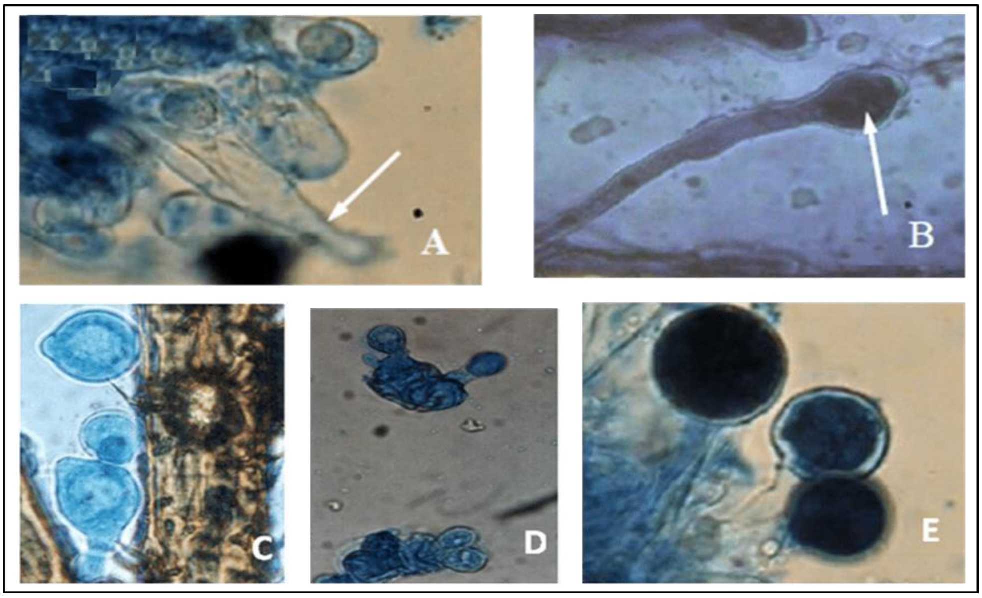

Fig. 12. Light micrographs showing *C. thromboides* infected *N. viridula* stink green bug: (A) Extended tips of Conidiogenous cells before conidia develop (arrow) on *N. viridula* X 400; (B) Developing conidia at apex of Conidiogenous cells and conidia X 400; (C) Fungus *C. thromboides* showing conidiophores ready to discharged primary conidium on *N. viridula* X 400 (Nada, 2006); (D) Globose conidia with hemispherical papilla attached with *R. padi* legs and (E) Germinated conidia on *R. padi* X 400 (Sewify unpublished data)

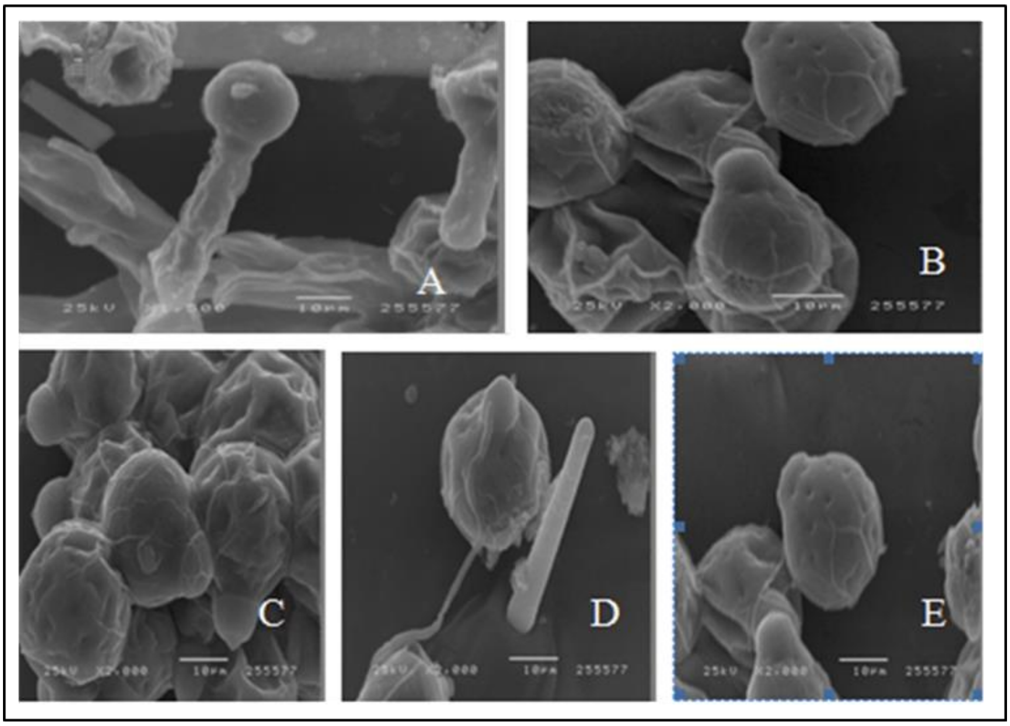

Fig. 13. Scanning electron microscopy showing *C. thromboides* infected *N. viridula* stink green bug (A) Conidia of *C. thromboides* in the process of formation on the conidiophores X 1500; (B) Conidiophore's of *C. thromboides* ready to discharged primary conidia X 2000; (C) Aggregation of the primary conidia of *C. thromboides* X 2000; and (D-E) Pyriform primary conidia discharged by papilla reversion X 2000 [23]

5. CONCLUSION

This review focuses on the geographical distribution and morphological description of various entomophthoralean species, attacking sucking insects in Egypt. Up till now ten Entomophthorales fungi species belonging to three families have been recorded from sucking insects, which have served as their host. These fungi distributed in several Governorates, representing Lower and Upper Egypt, acquiring a wide range of climatic conditions. The review shows the need to do more efforts to isolate and define the group of these fungi by using modern identification techniques. Also, more studies are needed on their epidemiology and ability to predict their occurrence under climatic conditions in Egypt.

AVAILABILITY OF DATA AND MATERIALS

All data generated and/or analyzed during the present study are available in the manuscript, and the corresponding author has no objection to the availability of data and materials.

DISCLAIMER (ARTIFICIAL INTELLIGENCE)

Author(s) hereby declare that NO generative AI technologies such as Large Language Models (ChatGPT, COPILOT, etc) and text-to-image generators have been used during writing or editing of manuscripts.

COMPETING INTERESTS

Authors have declared that no competing interests exist.

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