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# African dust is an unlikely source of *Aspergillus sydowii*, the causative agent of sea fan disease

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ABSTRACT: Infection of sea fans by the fungal pathogen *Aspergillus sydowii* is one of the most widespread coral diseases in the Caribbean. The source of this normally terrestrial fungus in marine ecosystems is perplexing, but of interest to coral conservationists, since tracking sources of pathogens provides one of the few avenues to limit pathogen spread. Hypothesized inputs of *A. sydowii* include terrestrial deposits, marine sources, and African dust. Windborne dust from Africa amounts to nearly $1 \times 10^9$ t yr$^{-1}$, much of which is deposited over the Caribbean region. Several studies have examined the microbiota of African dust and detected the presence of *Aspergillus* spp., although identifications were only to the genus level. I used specific culture conditions to determine whether this coral pathogen is present in 4 samples of airborne dust from the Caribbean and Africa, and 3 sediment samples from Africa and the Cape Verde Islands (eastern Atlantic). A diversity of fungi were found, including 7 species of *Aspergillus* and related taxa. However, none of the samples contained *A. sydowii*. The lack of *A. sydowii* in airborne dust and sediment samples suggests that African dust is an unlikely source of the marine pathogen *A. sydowii*. Given the high richness of fungi observed, even under selective growth conditions, identification of potential pathogens to the species level is critical.

KEY WORDS: Coral disease · Aspergillosis · African dust · Sahara · Long-distance transport · Sea fan disease



## INTRODUCTION

Coral reefs are an ecosystem increasingly suffering the synergistic effects of a variety of stressors, including temperature (Hoegh-Guldberg 1999, Harvell et al. 2002), over-fishing (Hughes 1994), eutrophication (Fabricius 2005), and disease (Harvell et al. 1999, Harvell et al. 2007). The dramatic rise in coral disease since the 1970s (Harvell et al. 1999, Ward & Lafferty 2004) is an area of considerable attention, as the causes behind the origin and spread of marine disease remain largely unknown. One factor that has been suggested to affect both the host and the pathogen is the input of wind-borne dust into Caribbean ecosystems (Shinn et al. 2000, Hayes et al. 2001, Garrison et al. 2003). The transport of dust from Africa to the Atlantic and Caribbean via trade winds has long been known by mariners (Gorbushina et al. 2007), and was first quantified in the late 1960s (Prospero 1968, Prospero et al. 1970). The amount of deposited dust is significant (ca. $1 \times 10^9$ t yr$^{-1}$, D'Almeida 1986), and has been increasing steadily since the 1970s (Prospero & Nees 1986, Prospero & Lamb 2003). Recent climate modeling indicates that this increase in dust transport is due to a complex interaction of factors including sea surface temperature, global climate, regional meteorology, and surface materials (Moulin et al. 1997, Giannini et al. 2003, Prospero & Lamb 2003, Moulin & Chiapello 2006).

The pattern of increased African dust since the 1970s correlates strongly with the observed increase in disease outbreaks in Caribbean coral reef communities (Hayes et al. 2001). There are several hypothesized mechanisms by which an increase in African dust

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could result in an increase in coral disease. African dust is known to enhance deposition of nutrients to the Atlantic (Prospero et al. 1996, Arimoto et al. 2003), including iron, a limiting micronutrient in many marine systems (Jickells et al. 2005). Local enrichment of iron as a result of dust deposition may facilitate pathogen growth and spread (Hayes et al. 2001) or cause declines in coral health, since corals normally live in oligotrophic waters. Besides nutrients, African dust also deposits organic pollutants (van Dijk & Guicherit 1999, Erel et al. 2006, Garrison et al. 2006), which may impair host immunity and lead to an increase in disease (Garrison et al. 2003, Garrison et al. 2006). Finally, African dust has been posited as a direct source of pathogenic organisms (Shinn et al. 2000). This hypothesis is consistent with numerous examples of long-distance dispersal (via wind) for a range of plant (Brown & Hovmoller 2002, Aylor 2003) and animal (Venkatesh et al. 1975, Griffin et al. 2001a) pathogens.

For African dust deposits to be directly responsible for increasing pathogen inputs, microorganisms must be able to survive the conditions of the high atmosphere (temperature, UV) for about 1 wk, the average transit time for a dust cloud to move from Africa to the Caribbean (Prospero & Nees 1986, Prospero et al. 2005). Despite the harsh conditions, a diversity of microorganisms have been detected in dust samples (Griffin et al. 2001b, Griffin et al. 2003, Shinn et al. 2003, Kellogg et al. 2004, Weir-Brush et al. 2004). The survival of these organisms is supported by models indicating that high altitude dust clouds can act as a highly efficient UV screen, protecting microorganisms in dust at lower altitudes (Herman et al. 1999). In addition, many of the microbes in dust clouds are pigmented (Garrison et al. 2003, Kellogg et al. 2004), which would offer additional protection from UV radiation.

African dust has been suggested as the source of *Aspergillus sydowii*, the pathogen responsible for aspergillosis in Caribbean gorgonian corals (Shinn et al. 2000). This disease was first observed in the 1980s (Garzón-Ferreira & Zea 1992), and has caused high rates of mortality throughout the Caribbean (Nagelkerken et al. 1997, Kim & Harvell 2004). This disease system has been a model for experimental studies as both the host and pathogen can be successfully grown in the laboratory. Despite the high prevalence and large effect of *A. sydowii* on gorgonian-dominated communities, the origin of the pathogen is unknown and hotly debated (Shinn et al. 2000). Some authors have suggested terrestrial sediments (Smith et al. 1996, Geiser et al. 1998) and marine sources (Roth et al. 1964, Steele 1967) as the most likely pathogen inputs. Shinn et al. (2000) cite the presence of the genus *Aspergillus* in dust as evidence that African dust is the primary source of the coral pathogen *A. sydowii.*

Several previous studies examined the fungal biota of African dust (Griffin et al. 2001b, Griffin et al. 2003, Shinn et al. 2003, Kellogg et al. 2004, Weir-Brush et al. 2004), and, although several isolated *Aspergillus* spp. (Griffin et al. 2003, Shinn et al. 2003, Kellogg et al. 2004, Weir-Brush et al. 2004), none specifically documented the presence of *A. sydowii*. Given that there are at least 180 recognized species within this genus (Pitt et al. 2000), and the high frequency of *Aspergillus* in soil samples (Domsch et al. 1980), odds are low that any given *Aspergillus* species would be *A. sydowii*. Despite this, numerous authors continue to cite support for the hypothesis that African dust deposits are the source of this coral pathogen (Shinn et al. 2000, Garrison et al. 2003, Weir-Brush et al. 2004). The lack of identified *A. sydowii* in dust samples may reflect either the genuine absence of this microbe in African dust, or may be the result of insufficiently precise culture techniques. Growth media and temperature can strongly influence microbes cultured from environmental samples. In addition, culture-based techniques are themselves biased in that they only identify a subset of organisms (Barer et al. 1993). Given that the spores of *Aspergillus* are robust to temperature and UV stress (Abdalla 1988, Klich 2002a), specific methods should be used to investigate whether *A. sydowii* is transported to the Caribbean in wind-blown dust.

To determine whether African dust is a source of *Aspergillus sydowii* in Caribbean coral reefs, I identified fungi cultured from airborne dust collected in the Caribbean and Africa, and sediment samples from Africa and the Cape Verde Islands (in the eastern Atlantic). I used specific culture techniques to select for *Aspergillus*, and identified fungi within the genus to the species level.

## MATERIALS AND METHODS

Airborne dust was collected from 2 locations (Table 1), viz. Emetteur Kati, Bamako, Mali (12° 41′ 17″ N, 8° 01′ 09″ E) at an elevation of 555 m during 3 dust events (March 2006), and Pt. Udall, St. Croix at an elevation of 40 m during a dust event (September 2006). In both locations, samples were taken using a portable vacuum filter sampling device described by Kellogg et al. (2004). Briefly, air was filtered through sterile cellulose nitrate membranes with a pore size of 0.2 µm, at a flow rate of 10 l $min^{-1}$ for 10 to 12 min. Following sampling, the filters were sealed, placed in Ziploc™ bags, and mailed to the USA. To account for potential contamination during handling, shipping, and processing of the filters, a control was included whereby the filtra-

Table 1. Airborne dust sampling locations and conditions, mean fungal colony forming units per liter of air sampled (CFU $l^{-1}$) following 14 d of growth at 25°C. SLPM: standard liter $min^{-1}$

| Sampling location | Date | Air temperature (°C) | Sampling duration (min) | Flow rate (SLPM) | Dust event | Wind speed (km $h^{-1}$) | CFU $l^{-1}$ |
|---|---|---|---|---|---|---|---|
| Emetteur Kati, Bamako, Mali | 23/3/2006 | 38 | 10 | 12.5 | Yes | 12 | 0.076 |
| Emetteur Kati, Bamako, Mali | 26/3/2006 | 40 | 10 | 12.5 | Little dust | 1–15 | 0.019 |
| Emetteur Kati, Bamako, Mali | 28/3/2006 | >40 | 10 | 12.25 | Smoke/dust | 5–10 | 0.043 |
| Pt. Udall, St. Croix | 17/9/2006 | 40.5 | 12 | 17.5 | Yes | 18–25 | 0.019 |

tion apparatus was removed from its sterile packaging, but the cellulose membrane was not directly exposed to air. These samples were otherwise handled in an identical manner to the membranes that had air filtered across them.

Upon arrival in Ithaca, NY, USA the filters were cut in half with sterile scissors, and placed sample side up on malt extract agar (MEA) and dichloran-glycerol agar (DG18), both amended with 50 µg $ml^{-1}$ tetracycline. These media were chosen specifically with the aim of culturing the coral pathogen *Asperigillus sydowii*. MEA is a commonly used medium for *Aspergillus* spp. (Klich 2002b), and best visualizes the diagnostic color of *A. sydowii*. DG18 is used to culture xerophilic fungi, such as *Aspergillus* spp. (Hocking & Pitt 1980). Plates were incubated at 25°C for 14 d. This incubation temperature is ideal for *A. sydowii* (Raper & Fennell 1965), and is similar to that of previous fungal isolations from dust samples (Griffin et al. 2001b, Griffin et al. 2003, Kellogg et al. 2004, Weir-Brush et al. 2004). During the 14 d incubation period, all fungal colonies were counted and sub-cultured onto MEA daily.

Samples of surface sediment were taken in March 2006 from 1 site in Africa (Bamako, Mali), and 2 sites on Sal Island, in the Cape Verde Islands (Pedre de Lume and Punta Fiura). Samples were taken from the top 0.5 cm of sediment over a surface area of approximately 55 $cm^2$, and were collected and packaged using sterile technique. Upon arrival in the USA, the sediment was serially diluted ($10^{-2}$, $10^{-3}$, $10^{-4}$, $10^{-5}$) and plated onto both MEA and DG18 amended with tetracycline. Plates were incubated at 25°C for 14 d. All fungal colonies were counted and sub-cultured daily.

Fungi were identified to genus using Domsch et al. (1980) and St. Germain & Summerbell (1996). All *Aspergillus* spp. were identified to species according to Klich (2002b); colony-level morphological characteristics (color, growth rate) and microscopic characteristics (standard 100× light microscopy for shape and color of conidia, sporulating structures (vesicles) and hyphae) were examined. Species identification is based on characteristic morphologies when the fungus is grown on 3 diagnostic media (Czapek yeast extract agar, Czapek yeast extract agar with 20% sucrose, and malt extract agar) at 2 temperatures (25°C, 37°C). All fungi were preserved in both 70% ethanol and phosphate buffered saline with 10% glycerol, and stored at −80°C. The number of fungal colony forming units per liter of air sampled (CFU $l^{-1}$) are reported for airborne dust, and the number of fungal colony forming units (CFU) are reported for sediment samples.

## RESULTS

Both culturing media (DG18 and MEA) were effective in isolating a diversity of fungi from the airborne dust samples (Table 2). After 2 wk of growth, the number of fungi in the samples from Mali ranged from 0 to 0.104 CFU $l^{-1}$, and those from St. Croix ranged from 0.005 to 0.033 CFU $l^{-1}$ (Table 2). Overall, samples from Mali had higher fungal abundances than those from St. Croix, although the 3 sampling dates in Mali were quite variable. Fungal abundance appears to be related to the magnitude of the dust event; the highest fungal counts were obtained during obvious dust events (Mali, 23 March), and the lowest when little dust was in the air (Mali, 26 March). No fungus grew in the handling controls, indicating that the manipulation, shipping, and storage of samples was effective in preventing contamination.

The most common fungal genus in airborne dust samples from Mali was *Cladosporium*, comprising 35.5% of all colonies (Table 2). In St. Croix, the most common fungal genus was *Acremonium*. Seven species of *Aspergillus* (including *Emericella* sp. and *Eurotium* sp., sexual stages of *Aspergillus* spp.) were observed overall, and ranged from 3 to 15% of the total colonies (Table 2). Airborne dust from Mali contained all 7 *Aspergillus* species, and dust from St. Croix contained only *A. niger*. None of the recorded *Aspergillus* colonies was *A. sydowii*. Although *Aspergillus* can be a taxonomically challenging genus, I have high confi-

Table 2. Fungal species isolated from airborne dust collected in Mali and St. Croix. Abundance is the proportion of colony forming units (CFUs) on both types of culturing media (DG18 and MEA) for all replicate filters following 14 d of growth at 25°C

| Fungal species | Abundance (% total CFU) | | | |
|---|---|---|---|---|
| | Mali | | | St. Croix |
| | 23 March | 26 March | 28 March | 17 Sept |
| *Cladosporium* spp. | 49.1 | 0 | 28.1 | 6.3 |
| Yeast | 6.8 | 62.5 | 0 | 9.4 |
| All *Aspergillus* spp. | 10.2 | 6.3 | 15.6 | 3.1 |
| *A. fumigatus* | 0 | 0 | 3.1 | 0 |
| *A. niger* | 3.4 | 0 | 6.3 | 3.1 |
| *A. niveus* | 0 | 6.3 | 0 | 0 |
| *A. sydowii* | 0 | 0 | 0 | 0 |
| *A. terreus* | 3.4 | 0 | 0 | 0 |
| *A. ustus* | 0 | 0 | 3.1 | 0 |
| *Eurotium* sp. | 1.7 | 0 | 0 | 0 |
| *Emericella* sp. | 0 | 0 | 3.1 | 0 |
| *Penicillium* spp. | 1.7 | 0 | 3.1 | 3.1 |
| *Curvularia* spp. | 1.7 | 0 | 6.3 | 0 |
| *Acremonium* spp. | 0 | 0 | 3.1 | 15.6 |
| *Fusarium* spp. | 0 | 0 | 0 | 9.4 |
| *Stachybotrys* sp. | 0 | 6.3 | 0 | 0 |
| *Paecilomyces* sp. | 0 | 0 | 0 | 3.1 |
| *Sporothrix* sp. | 0 | 0 | 0 | 3.1 |
| *Nigrosporium* sp. | 0 | 0 | 0 | 3.1 |
| Unidentified filamentous fungi | 30.5 | 25 | 43.8 | 43.8 |

dence in the species identifications as I used an established set of morphological characters based on growth on 3 types of diagnostic media at 2 temperatures (Klich 2002b). Of the species observed, none was easily confused with *A. sydowii*, which has a characteristic turquoise colony color, relatively slow colony growth rates, and rough-walled conidia (Klich 2002b).

Table 3. Fungal species from sediment samples collected in Mali (26 March 2006) and Cape Verde (16 March 2006). Abundance is the number of colony-forming units (CFUs) averaged over both types of culturing media (DG18 and MEA) for all replicate filters following 48 h of growth at 25°C

| Sediment dilution | Abundance (CFU) | | |
|---|---|---|---|
| | Mali | Cape Verde | |
| | Bamako | Pedra de Lume | Punta Fiura |
| $10^{-2}$ | 58.5 | 25.5 | 5.5 |
| $10^{-3}$ | 30 | 7.5 | 0 |
| $10^{-4}$ | 9 | 1 | 0 |
| $10^{-5}$ | 4.5 | Overgrown | 0 |
| | Common fungal species | | |
| | Bamako | Pedra de Lume | Punta Fiura |
| | *Aspergillus niger*, *Cladosporium* spp., zygomycete, *Penicillium* spp., yeast | *Cladosporium* spp., *Aspergillus ustus*, *A. terreus*, yeast, zygomycete | *Cladosporium* spp. |

Sediment samples had much higher fungal abundances than air samples, and a dilution of at least $10^{-4}$ was necessary to prevent complete overgrowth within 48 h (Table 3). Samples from one of the sites on Cape Verde (Punta Fiura) had low abundance of fungi; dilutions greater than $10^{-3}$ showed no growth. Overall, it was difficult to assess fungal diversity in the sediment samples due to the high density of colonies. Species observed were similar to those in the airborne dust samples (*Cladosporium*, *Penicillium*, yeast, *Aspergillus* spp.). Three species of *Aspergillus* were observed (*A. niger*, *A. terreus*, *A. ustus*), but *A. sydowii* was not among them (Table 3).

## DISCUSSION

No evidence of the coral pathogen *Aspergillus sydowii* was found in 3 airborne dust samples from Africa and 1 from the Caribbean, or in sediment samples from Africa and the Cape Verde Islands (Tables 2 & 3). Our targeted approach using culturing media selective for the growth of *Aspergillus* (MEA and DG18) and detailed identification methods (Klich 2002b) was successful, yielding 7 different species of *Aspergillus* and related taxa. The most common taxon was *Cladosporium* spp., which agrees with Lacey's (1991) observation that this is the most abundant fungus in temperate and tropical air samples.

A comparison of these results with previous examinations of the fungal biota from African dust revealed some interesting trends (Table 4). The abundance of fungi in Mali air samples from this study were lower than those collected in 2001 and 2002 in the same region (Kellogg et al. 2004). This could be the result of factors such as large-scale climate patterns (i.e. North Atlantic Oscillation) or local conditions (humidity, UV, temperature, aerosol concentrations). Temporal variation in microbial abundance in air samples is well documented, and is related to the concentration of dust in the air, which varies seasonally, with maximum deposition in June/July, and inter-annually, with deposition strongly tied to low rainfall in Africa and large-scale climate processes (Nickling & Gillies 1993, Fontaine & Janicot 1996, Tegen et al. 1996).

From an examination of previous studies, it was not possible for me to assess the role of different culture techniques on fungal abundance, as this was confounded with both sampling location and time (Table 4). However,

Table 4. Comparison of methodology and results of this and previous studies examining the fungal biota of African dust. Only samples taken during documented dust events are included. CFU $l^{-1}$: colony forming units per unit of filtered air. Y: yes, N: no, USVI: US Virgin Islands

| Sampling | | Media | Incubation | | CFU $l^{-1}$ | Presence of | | Source[a] |
|---|---|---|---|---|---|---|---|---|
| Location | Date | | Temp. (°C) | Time | | *Aspergillus* | *A. sydowii* | |
| St. John | Jul 2000 | R2A | Room | 2 wk | 0.048 | N | N | 1 |
| USVI | Jul 2001 | R2A | 23 | 48 h | 0.024 | N | N | 2 |
| USVI | Aug 2001 | R2A | 23 | 48 h | 0.065 | Y | Unknown (to genus) | 2 |
| USVI | Jul 1999 | YEG, MEG | 28 | Unknown | – | Y | Unknown (to genus) | 3 |
| USVI | Sept 1999 | YEG, MEG | 28 | Unknown | – | Y | Unknown (to genus) | 3 |
| Mali | 2001, 2002 | R2A | 26 | 48 h | 0.225 | Y | Unknown (to genus) | 4 |
| Barbados | Apr 1996 | Sabouraud | 37 for 48h, 30 for 2 wk | 2 wk | 0.003 | – | – | 5 |
| Barbados | Jun 1997 | Sabouraud | 37 for 48h, 30 for 2 wk | 2 wk | 0.015 | – | – | 5 |
| Mali | Mar 2006 | MEA, DG18 | 25 | 2 wk | 0.035 | Y | N | 6 |
| St. Croix | Sept 2006 | MEA, DG18 | 25 | 2 wk | 0.015 | Y | N | 6 |

[a]1: Griffin et al. (2001b), 2: Griffin et al. (2003), 3: Weir-Brush et al. (2004), 4: Kellog et al. (2004), 5: Prospero et al. (2005), 6: This study

several authors found no significant difference in CFUs using nutrient-rich (i.e. Sabouraud) and nutrient-poor (i.e. R2A) media (Kellogg et al. 2004, Prospero et al. 2005). I also found little difference in either fungal abundance or diversity between the 2 types of culturing media used (DG18 and MEA). Some of the lowest fungal abundances observed were from Barbados (Prospero et al. 2005) in a study that used the highest incubation temperature (30°C). This might be the result of either the temperature under which fungi were grown, or geographic variation.

Several previous studies have documented the presence of *Aspergillus* spp. in African dust (Griffin et al. 2003, Kellogg et al. 2004, Weir-Brush et al. 2004). However, none of these identified fungi to the species level. Methods of identifying fungi from dust samples include morphology (Weir-Brush et al. 2004) and 16S/18S rDNA sequencing (Griffin et al. 2001b, Griffin et al. 2003, Kellogg et al. 2004). The presence of *Aspergillus* spp. in samples does not necessarily indicate that the coral pathogen *A. sydowii* is present; this is a diverse fungal genus and species identification can be difficult. For example, *A. versicolor*, which is a morphologically similar close relative of *A. sydowii*, is extremely widespread in soil and air samples (Domsch et al. 1980, Klich 2002a, Gorbushina et al. 2007). I used detailed morphological analysis of fungi based on growth on 3 types of diagnostic media at 2 temperatures (Klich 2002b) to confirm the identity of all *Aspergillus* species. This method has high precision at the species level, in contrast to 16S/18S rDNA sequencing used by previous studies that can only identify fungi to genus. Recently developed species-specific molecular probes (Haughland et al. 2004) may provide an alternate approach to identifying fungi at higher taxonomic resolution in dust samples. Regardless of the method, if the goal is to identify specific fungi (such as the coral pathogen *A. sydowii*), more detailed analyses of the morphological and molecular traits are required to assign species identity.

It is important to realize that the presence of viable fungus in African dust samples does not necessarily indicate that it is capable of infecting coral hosts. Likewise, studies such as mine may overlook non-culturable isolates of fungus. The environmental conditions a fungus encounters prior to infecting a coral host are not easily duplicated in the laboratory, and therefore it is possible that not all fungi capable of infecting hosts are cultured using traditional methods. However, given the resilience of fungal spores to the harsh conditions found in dust clouds, and the optimal nutrient and temperature conditions of the culturing media, it seems likely that most viable spores would germinate. Whether these spores are capable of infecting coral hosts requires further study.

A lack of *Aspergillus sydowii* in dust samples from both Africa and the Caribbean in this study makes it tempting to conclude that African dust is not a viable source of this coral pathogen. However, given the large spatial and temporal variation in fungal diversity and abundance, I cannot conclusively rule out the African Dust hypothesis, or the possibility that *A. sydowii* was present in dust from earlier years. Comparisons with previous studies emphasize that inputs from African dust into marine systems are likely to be spatially and temporally variable, and specific culturing and identification methods must be used to conclusively determine the presence of *A. sydowii* in African dust. Future tests of the African Dust hypothesis are important and should use targeted searches with potential pathogens identified to as fine a taxonomic

resolution as possible. It is not sufficient to identify to the genus level, as aspergillosis of corals is caused by a single species. *Aspergillus* is a diverse genus, and the evidence of several other species of *Aspergillus* within our dust samples suggests that taxonomic resolution to the species-level is necessary.

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