Soiled adhesive pads shear clean by slipping: a robust self-cleaning mechanism in climbing beetles

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Animals using adhesive pads to climb smooth surfaces face the problem of keeping their pads clean and functional. Here, a self-cleaning mechanism is proposed whereby soiled feet would slip on the surface due to a lack of adhesion but shed particles in return. Our study offers an in situ quantification of self-cleaning performance in fibrillar adhesives, using the dock beetle as a model organism. After beetles soiled their pads by stepping into patches of spherical beads, we found that their gait was significantly affected. Specifically, soiled pads slipped 10 times further than clean pads, with more particles deposited for longer slips. Like previous studies, we found that particle size affected cleaning performance. Large (45 \( \mu \)m) beads were removed most effectively, followed by medium (10 \( \mu \)m) and small (1 \( \mu \)m). Consistent with our results from climbing beetles, force measurements on freshly severed legs revealed larger detachment forces of medium particles from adhesive pads compared to a flat surface, possibly due to interlocking between fibres. By contrast, dock leaves showed an overall larger affinity to the beads and thus reduced the need for cleaning. Self-cleaning through slippage provides a mechanism robust to particle size and may inspire solutions for artificial adhesives.

1. Introduction

Many animals, including insects, spiders, geckos and tree frogs, can climb smooth surfaces by using adhesive pads on their feet. Morphologically, these pads are either hairy or smooth. Hairy pads exhibit a brush-like array of thin fibres (setae) and are common among spiders and geckos, but are also found in insects like flies and beetles. Smooth pads lack such hairs and are found in many insects, like ants, cockroaches and stick insects, as well as vertebrates like tree frogs. As both types of pads are inherently sticky, they attract dirt particles. Without cleaning, the pads would foul quickly and become unusable. While there may be several mechanisms used for cleaning, like grooming [1], brushing with other legs [2] or flushing with fluids [3,4], these mechanisms are time or energy consuming, and so would greatly impede locomotory performance. For these reasons, a self-cleaning mechanism was proposed whereby the particles would be brushed or rolled off through interactions between the pad and substrate. Such a passive mechanism was first shown by Hansen & Autumn for geckos [5] and later in several insects, including beetles and stick insects [3] and ants [4,6].

The exact nature of this biological cleaning mechanism, commonly referred to as contact self-cleaning, still remains unclear. Hu and co-authors proposed that the adhesive hairs in geckos could flick off particles when the hairs spring back to their default position after detachment [7]. Xu and co-authors showed that the tuning of detachment speed can be a possible cleaning mechanism, with particles being deposited at higher detachment speeds [8]. Further aspects on the rate of cleaning include the geometry of the fouling particles (like shape [6] and size [3,6]) and the surface chemistry of the particles relative to the substrate [4]. Apart from the properties of the particles
themselves, Clemente and co-authors showed that the two
types of adhesive pads found in insects (i.e. smooth and
hairy pads) exhibited differences in self-cleaning ability.
Hairy pads were more efficient in self-cleaning compared
with smooth pads, except for particles that are in a size
range where they would be lodged between the adhesive
hairs [3]. Additionally, the authors found that the adhesive
stress, or adhesion with respect to the contact area, is constant
and that soiling decreases adhesion by decreasing the contact
area. Therefore, recovery of adhesion correlates with the
removal of particles. The above-mentioned aspects were
also verified by testing artificial, fibrillar mimics. The studies
investigated the roles of fibre stiffness [9], pull-off speed [8]
and inter-setal distance [10,11]. Despite these efforts, many
artificial adhesive pads still suffer from fouling.

Many of the above-mentioned studies either tested
restrained animals or artificial mimics. As it was shown by
Crawford and co-authors, the rate of self-cleaning in freely
climbing tree frogs is greatly improved over simulated steps
in restrained frogs [12]. A possible effect could be that in
unrestrained animals the pads slip for a longer distance or
more often, and in return clean faster. In this study, we aim
to test this hypothesis by recording the in situ self-cleaning
behaviour of freely climbing beetles and relate them to the
leg kinematics.

In this work, we address the following questions: (i) do
beetles clean their pads faster when unrestrained? (ii) Do bee-
tles show different leg movements, like increased slippage,
when their pads are soiled? (iii) Are there any lessons we
can translate to synthetic adhesive systems?

2. Methods

2.1. Model organism

Our model organism is the male green dock beetle (Gastrophysa
viridula) shown in figure 1. This species has been widely studied
as a model for biological, fibrillar adhesive pads [3,13–17]. The
fibrillar adhesive pads of the beetles, shown in figure 2a, have
fibres that are 40 μm long, 2 μm thick, and have a 10-μm gap
between them, as well as three different tip geometries (discoid-
al, spatula and pointed) with varying material and adhesive
properties [3,13]. They are also of interest for engineered
systems because their geometry closely resemble synthetic,
mushroom-tipped, fibrillar adhesives [18–26].

We procured dock beetles from the forests around the Stuttgart-
Vaihingen and Würzburg (Germany) regions. A colony was
maintained in the laboratory and fed wild-picked dock leaves
(Rumex obtusifolius). For our climbing observations, we chose
10 individual male beetles of a similar weight (m = 10 ± 0.5 mg).

2.2. Soiling of substrates

The self-cleaning abilities of beetles were tested on two different
substrates, flat glass and the top side of a dock leaf, with three
different sizes of spherical particles (1 μm, 10 μm and 45 μm
in diameter). The particle sizes were chosen following previous
research on self-cleaning of beetle pads [3]. In particular, the
1-μm particles are much smaller than the space between the
fibres (approx. 10 μm; figure 2b,c) and easily penetrate into
the fibre arrays, the 10-μm particles fit between the fibres
(figure 2d,e), and the 45-μm particles are larger than the gaps
and cannot penetrate into the fibre arrays (figure 2f,g). We used
untreated polystyrene microbeads (Polysciences, Inc., Washington,
PA, USA; 0.99 ± 0.03 μm, 10.0 ± 0.56 μm and 49.1 ± 1.34 μm) for

the trials on the glass substrates. For the trials on the dock leaf
substrate, we used polystyrene microbeads embedded with a
fluorochrome dye that excites at a wavelength of 530 nm and
emits at 607 nm for fluorescence imaging (microParticles,
GmbH, Berlin-Aldershof, Germany; 10.2 ± 0.13 μm and 48.2 ±
0.60 μm), as fluorescence was required to identify the beads on
the reflection images taken of the leaves.

To deposit the particles onto the glass substrate, we used a
technique similar to that reported in a previous study [3]. The
particles were cleaned of surfactants and suspended in water.
The glass substrates were rendered hydrophilic through oxygen
plasma treating (Diener Electronic Zepto, Ebhausen, Germany)
in order to allow spreading of the aqueous particle solutions.
A 5-μl drop of the aqueous particle solution was deposited onto
the treated glass, flash frozen with liquid nitrogen, and
then the water was sublimated using a freeze dryer (Dieter Piat-
kowski Forschungsgerate P4K Freeze Drier with JUMO IMAGO
F3000 controller unit, Petershausen, Germany). This process
ensured the deposition of a uniform monolayer of particles by
getting rid of the capillary effects from the liquid water; thus,
preventing the accumulation of particles into thin rings—
commonly referred to as ‘coffee rings’ [27]. Figure 1 shows a
glass surface with 45-μm microbeads deposited.

To avoid damaging the leaf surface, freeze drying was not an
option for the dock leaf surface. Instead, the surfactant-free aq-
ueous particle solution was deposited onto the leaf surface and left
for the water to evaporate. This process resulted in more or less
uniform monolayer depositions of the particles. Unlike the glass
surface, the water of the particle solution was absorbed by the

Figure 1. Set-up for contaminating the adhesive pads in climbing dock beetles. Male dock beetles (Gastrophysa viridula) were allowed to climb vertically up on a clean glass surface after stepping into a patch of microbeads. The rectangles represent the areas around each foot placement (here the left foreleg) where we counted accumulated and deposited beads. The arrow represents the direction of gravity. Inset: a beetle on its natural substrate, a dock leaf (Rumex obtusifolius). Scale bars represent 5 mm and 1 mm (inset). (Online version in colour.)
The increased water loss due to absorption may have helped overcome the capillary effects, also preventing the formation of ‘coffee rings’ [28].

2.3. Gait kinematics and self-cleaning in climbing beetles

An individual beetle was allowed to climb vertically on either a clean glass microscopy slide (25 × 70 × 1.0 mm), or a dock leaf mounted on the glass slide with double-sided adhesive tape. The glass substrate had patches of either 1-μm, 10-μm or 45-μm particles, while the leaf substrate had patches of 10-μm or 45-μm particles. The patches were located towards the bottom half of the substrate to allow the beetle to step into the patch and climb on the clean substrate, as shown in figure 1.

The climbing behaviour of the beetle was filmed from the dorsal side using a high-speed camera (Vision Research Phantom v641, Wayne, NJ, USA) with a Nikon 24-85 mm f/2.8-4D AF Nikkor lens. The videos were captured at a resolution of 1920 by 1080 pixels and 250 frames per second, which allowed us to distinguish well the timing and position of individual steps.

The position of the distal portion of a leg (shown in figure 2a) was tracked using an open source tracking software (Tracker by Douglas Brown, http://physlets.org/tracker/). From the tracking data, we were able to extract the footfall position of individual legs throughout the climb. In order to capture the span of foot slippage, we defined rectangular regions of interest by adding and subtracting the distal–proximal length of the last tarsal segment to the maximum and minimum locations of the footfall, respectively. Figure 1 shows an example of the regions of interest for the left foreleg overlaid on a screenshot from one of the videos (see also the electronic supplementary material, video SV1). The experiments were conducted in a climate controlled room with a temperature of 23°C and 30% relative humidity.

In order to facilitate counting particles being picked up and deposited by the beetle, a very high-resolution image (54208 by 18542 pixels) of the entire slide was taken before and after a beetle climbed on it. The image was captured using an inverted microscope (Nikon Eclipse Ti, Düsseldorf, Germany) with a motorized stage (Marzhauser Wetzlar TANGO Desktop, Wetzlar, Germany) and a Hamamatsu ORCA-Flash4.0 V2 Digital CMOS (Hamamatsu, Japan) camera, in order to stitch together images with a 4× magnification objective. These high-resolution images provided a resolution of approximately 1 μm per pixel.

For the trials with the glass substrate, a uniform LED backlight was used to project silhouettes of the deposited particles. For the trials with the leaf substrate, a 542-nm laser was used to excite the particles, and their fluorescence emission was captured using a TRITC (590 ± 650 nm) filter.

Using MATLAB (MathWorks Inc., USA), we thresholded the images to black and white and subtracted the before and after images from each other to identify the particles picked up and deposited by the climbing beetle. The locations of the regions of interest where the beetle had stepped were then used to count the number of particles picked up and deposited by each step. This process was conducted with eight individual beetles for the glass substrate and two individuals for the leaf substrate.

In addition to the number of particles transferred to and from the substrate, we used the tracking data from the high-speed videos to analyse the gait kinematics. The particular kinematics we investigated were length of slip, frequency of the steps and duty cycle. The slip length corresponds to the shearing of the footpads while making contact with the substrate. The frequency of the steps corresponds to the number of steps taken per second.
The duty cycle corresponds to the ratio between the time in contact with the substrate (or the stance phase) and the total time of the step (stance and swing phase combined) [29]. To disregard resting steps, we only included steps that made contact with the substrate for less than 1 s.

High-speed and close-up observations of the removal of microbeads were conducted using an upright microscope (Zeiss Axio Imager.M2, Jena, Germany) and high-speed camera (Vision Research Phantom v641, Wayne, NJ, USA). The beetles were observed climbing upside down on a glass coverslip (24 × 60 × 0.15 mm) with microbeads deposited as previously described. We used objectives with 20× and 50× magnification, and captured the videos at a frame rate of 2000 frames per second. The videos are shown in the electronic supplementary material, video SV2.

2.4. Scanning electron microscopy

Scanning electron microscopy (SEM) was used to observe the adhesive pads of the beetles. Severed legs were prepared for SEM by fixing them to plastic studs (Baltic Präparation e.K., Niesgau, Germany) using conductive double-sided tape. They were coated in a 20-nm layer of gold using a sputter coater (Leica EM ACE600, Wetzlar, Germany). A Carl Zeiss Gemini (Jena, Germany) microscope was used with an electron acceleration of 0.5 kV.

We also used SEM to observe where the microbeads accumulated on the adhesive pads of beetles after taking 10 steps. Three male beetles were placed on prepared glass slides with 1-μm, 10 μm and 45-μm microbeads, respectively. They were observed to step into the patches of microbeads and then take 10 steps on the clean glass substrate. After the 10 steps, each beetle was picked up and immediately flash frozen using liquid nitrogen. The soiled foreleg was clipped and prepared for observation with SEM.

2.5. Atomic force microscopy

We used atomic force microscopy (AFM; JPK Instruments NanoWizard 4, Berlin, Germany) to directly measure the adhesion between 1-μm, 10-μm and 45-μm microbeads and the distal fibrillar pad of a beetle, the leaf substrate and the glass substrate. We used cantilevers with bending stiffnesses of 3–4 N m⁻¹ (NanoAndMore TL-FM-10 and ARROW-FMR-10, Wetzlar, Germany), following methods used in a previous study [30]. The microbeads were attached to the bare cantilevers using a two-part epoxy. For the measurements, we used a preload of 200 nN and pull-off velocity of 10 μm s⁻¹, which closely correspond to the parameters used in a previous study of the same beetle species [13]. The measurements were taken in a climate-controlled room with a temperature of 23°C and 30% relative humidity.

We tested adhesion on freshly ablated adhesive pads to ensure that we also captured possible effects arising from the adhesive fluid present on each adhesive hair. The adhesive fluid in insects is oily and evaporates only very slowly [31]. The fore- and midlegs from live, male dock beetles (N = 6) were ablated using precision scissors, and then mounted to a glass coverslip using a small volume of superglue. The pads were mounted with their adhesive hairs oriented vertically, similar to the orientation shown in figure 2a. The measurements were then taken within 30 min of preparation in order to minimize the evaporation of the adhesive fluid and desiccation of the fibres. For each pad, we took measurements at four different locations.

Similarly, we tested adhesion on the top surface of fresh dock leaf (N = 6). The leaves were collected from local forests and mounted on glass coverslips using double-sided tape. For each leaf, we took measurements at three different locations.

2.6. Contact angle measurements and surface characterization

We used a contact angle goniometer (Krüss Drop Shape Analyzer 100, Hamburg, Germany) to characterize the wettability of the glass and dock leaf substrates. Contact angles were measured using 2-μl drops of water and diiodo-methane. With the contact angles of both fluids, we were able to calculate the surface free energy. We used the Owens, Wendt, Rabel and Kaelble (OWRK) model for calculation as it accounts for the polar and dispersive parts of surface energy [32–34].

We scanned the topology of the dock leaf surface using a laser scanning microscope (Keyence VK-X200, Neu-Isernburg, Germany). The laser scans were conducted using an objective with 100× magnification.

3. Results

3.1. Particles removed during climbing

We observed 10 male dock beetles (Gastrophyta viridula) climbing on vertical glass (N = 8) and dock leaf (Rumex obtusifolius, N = 2) substrates. Figure 3a shows that the majority of the small (1 μm) microbeads were deposited during the first step. The subsequent steps removed very few microbeads. This is in contrast to the medium (10 μm) microbeads, which were constantly being deposited, with only a slight decrease in deposition for later steps (figure 3b). In figure 3c, we see that the large (45 μm) microbeads were deposited the most by the first step, followed by a gradual decrease in deposited beads with later steps. For all three bead sizes, we observed a monotonically decreasing trend for beads deposited and number of steps. We used Page’s L tests to determine the significance and found that all bead sizes follow this trend (L₁ = 10 600 with N = 32, L₁₀ = 9240 with N = 27, L₁₅₀ = 11 200 with N = 33, all with p < 0.001) [35].

By fitting an exponential decay to the median values, we were able to quantitatively compare the self-cleaning rates for each bead size. The exponential decay follows $N_p = N_p^0 e^{-b L}$, where $N_p$ is the number of particles (or beads) deposited and $N_p^0$ is the step number. The fitting coefficients a and b are tabulated in table 1. The exponential fits accurately represent the data, with the coefficients of determination $R^2$ varying between 0.60 and 0.73. The coefficient b represents the decay rate of the beads removed per step, with larger values corresponding to faster decays. We found that the removal of beads decays fastest for small beads, followed by large beads, and finally the medium beads.

Interestingly, when beetles climbed the leaf substrate almost no beads were deposited. In two trials, we found a single instance of a medium bead being deposited within 10 steps, and only six of the large beads.

3.2. Particles remaining on the adhesive pads

By comparing the number of microbeads picked up and deposited by the adhesive pads, we further quantified the effectiveness of self-cleaning by the climbing beetles. In figure 3d–f, we show the percentage of microbeads deposited for the small, medium and large microbeads, respectively. The large microbeads (figure 3f) were deposited more efficiently, with the deposition tapering off for the later steps. The first five steps removed 30% of the accumulated microbeads, while the final five steps removed another
On the other hand, the medium microbeads (figure 3e) were deposited at a constant rate. The small microbeads (figure 3d) were removed at the lowest rates, with the deposition also tapering off for the later steps. After 10 steps, the beetles removed 10% of the small microbeads, 20% of the medium microbeads and 40% of the large microbeads accumulated.

Figure 2b–g shows scanning electron micrographs of the adhesive pads of the beetles after being soiled and taking 10 steps. In figure 2b–c, we see that the small microbeads accumulated on the spatula-shaped or discoidal tips of the adhesive fibres, and were not effectively removed after the 10 steps. In figure 2d–e, we see that the adhesive pads were still covered with the medium microbeads. Specifically, as
compared to 2.5 mm, respectively.

Figure 4. Slipping increases with soiling on glass and leaves. The slip length for clean and soiled footpads on glass and leaf substrates.

shown in figure 2e, the medium microbeads were lodged between the adhesive fibres, and often stuck below the adhesive tips. In figure 2f–g, we see that the adhesive pads have only two of the large microbeads still attached. They were attached to the sides of the adhesive pads, where pointed-tip fibres are present.

3.3. Gait kinematics

With our observations of beetles climbing, we were able to determine how gait kinematics changed in situ when soiled. Overall, as shown in figure 4, the footpads slipped significantly further compared to clean pads (Kruskal–Wallis ANOVA: $\chi^2_{2,274} = 112.8, p < 0.001, N = 4$). During a natural, unsoiled climbing gait, beetle footpads slipped 0.12 mm (0.078 and 0.18 mm; median and interquartile ranges) on glass and 0.11 mm (0.079 and 0.16 mm) on leaves; however, soiled pads slipped 0.93 mm (0.55 and 1.3 mm) on glass and 0.32 mm (0.24 and 0.42 mm) on leaves. The typical slippage of a soiled footpad can be also seen in the electronic supplementary material, video SV1.

In figure 5a–c, we show how the slip length of the footpad affected the number of particles deposited on the glass substrate. For both small (figure 5a) and large (figure 5c) microbeads, longer slip lengths deposited greater numbers of particles. However, for medium (figure 5b) microbeads, slip lengths longer than 1.5 mm did not guarantee more particles deposited. The beetle pads soiled with medium microbeads also slipped greater lengths than the small and large microbeads, with maximum slip lengths of 4.5 mm compared to 2.5 mm, respectively.

In figure 5d–f, we show how the slip length changes with the number of steps taken after soiling. For all bead sizes, we observed decreases in slip length as the beetles climbed (Page’s L tests: $L_1 = 10800$ with $N = 32$, $L_{10} = 9190$ with $N = 27$, $L_{45} = 11100$ with $N = 33$, all with $p < 0.001$ [35]). However, the rates at which the slip lengths decreased varied. To quantify the rates, we fitted linear regressions through the median values, represented by the solid lines in figure 5d–f. The linear decay follows $L_x = cN_x + d$, where $L_x$ is the slip length and $N_x$ is the step number. The fitting coefficients $c$ and $d$ are tabulated in table 2. The linear fits accurately represent the data, with the coefficients of determination $R^2$ varying between 0.60 and 0.90. The coefficient $d$ represents the decay rate of the slip lengths per step, with larger values corresponding to faster decays. We found that the slip length decays fastest for large beads, followed by medium beads and small beads.

In addition to slip length, the step frequency and duty cycle were affected by soiling. In figure 6a, we show the frequency of steps for soiled and clean pads. Beetles with clean pads stepped at a significantly higher frequency than those with soiled pads (median value of 5.2 Hz compared to 2.9 Hz; for 1 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,280} = 79.96, p < 0.001, N = 8$; for 10 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,220} = 55.43, p < 0.001, N = 8$; for 45 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,280} = 65.42, p < 0.001, N = 8$). In figure 6b, we show the duty cycle, or ratio between the duration of the stance phase and the total step duration [29]. Beetles with cleaned pads spent around 70% of their stepping time in contact with the substrate, while beetles with soiled pads spent 80% of their stepping time in contact with the substrate. The beetles with cleaned pads spent significantly less time in contact with the substrate (for 1 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,280} = 64.41, p < 0.001, N = 8$; for 10 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,220} = 71.02, p < 0.001, N = 8$; for 45 μm, Kruskal–Wallis ANOVA: $\chi^2_{1,280} = 90.66, p < 0.001, N = 8$).

3.4. Effect of particle size on adhesion to footpads and substrates

Using AFM, we investigated the adhesive force between small, medium and large microbeads and the adhesive pad of a beetle, a glass substrate and a dock leaf substrate. In figure 7a, we show the adhesive forces between the three different bead sizes and various substrates. Statistical results are listed in tables 3 and 4.

From table 3, we found no significant difference between the three bead sizes when adhering to glass or leaf substrates (Kruskal–Wallis test: $p > 0.05$). However, there was a difference in adhesion to the adhesive pad of a beetle between the small and large, and medium and large beads.
There was no significant difference between the small and medium beads on the adhesive pad (Kruskal–Wallis test: $p < 0.05$). On average, the adhesive force between the medium beads and the adhesive pad was 2.5 times higher than the large beads.

We used our measurements to determine whether beads have stronger affinities towards substrates, tabulated in table 4. For the small and medium beads, we found that they have a significantly stronger affinity towards the adhesive pad than the glass substrate (Kruskal–Wallis test: $p = 0.11$ and $p < 0.05$, respectively). However, there was no significant difference between adhesion on the leaf or pad (Kruskal–Wallis test: $p > 0.05$). For the large beads, we found that they have a significantly stronger affinity towards the leaf substrate than the adhesive pad or glass substrate (Kruskal–Wallis test: $p < 0.05$). However, there was no significant difference between adhesion on the glass or pad (Kruskal–Wallis test: $p > 0.05$).

Wettability and surface free energy were also investigated as they are related to adhesion. As shown in the electronic supplementary material, table S1, the contact angles for

(Figure 5. Adhesive pad slipping promotes self-cleaning. (a–c) Relationship between number of beads removed and slip length of climbing beetles for (a) 1-µm, (b) 10-µm and (c) 45-µm polystyrene beads. Solid lines represent average number of beads removed for slip lengths between 0–0.5 mm, 0.5–1 mm, 1–1.5 mm and greater than 1.5 mm. (d–f) Slip length per step for climbing beetles soiled by (a) 1-µm, (b) 10-µm and (c) 45-µm polystyrene beads. Solid lines represent best linear fits, outlined in table 2. Dashed lines represent median slip length of unsoiled beetle pads. (Online version in colour.)
water drops on glass and dock leaf were found to be $8.1 \pm 1.0$ ($N = 9$) and $49 \pm 9.7$ ($N = 20$) degrees, respectively. The surface free energy for glass and dock leaf were found to be $74 \pm 1.4$ ($N = 9$) and $54 \pm 9.0$ mN m$^{-1}$ ($N = 20$), respectively. These values are reported as mean $\pm$ standard deviation.

### 4. Discussion

#### 4.1. Effect of particle size on self-cleaning

By accounting for the number of particles being picked up during soiling steps, we were able to quantify the relative deposition of particles in the cleaning process. Efficient contact self-cleaning was only observed for large (45 µm) particles. However, for medium particles (10 µm), we observed drastic decreases in self-cleaning performance, presumably because their size allows them to fit between the fibres. Only 20% of medium particles were removed after 10 steps, while 40% of the large particles were removed. Additionally, the few large particles remaining after 10 steps were found attached to the edges of the adhesive pads (see figure 2f–g and the electronic supplementary material, supporting video SV2). By aggregating on the edges, these large particles would not obstruct the adhesive part of the fibres and thus not affect adhesion and climbing performance.

The self-cleaning performance of small (1 µm) particles was found to be the lowest. This is consistent with the lower slip length (average 0.75 mm) compared to steps when soiled with the other bead sizes (average 0.89 mm for medium and 0.97 mm for large particles). As beetles were less affected by this particle size, climbing speed also was found to be slightly faster. After 10 steps, beetles soiled with small particles travelled an average distance of 14 mm, while those soiled with medium and large particles travelled 11 and 12 mm, respectively. The small particles also had a higher affinity to the adhesive pads than the glass, which supports the low rates of self-cleaning. The adhesive fluid underneath the tips of adhesive pads may be as thick as 0.5 µm [16,36]. While this is not enough to form capillary bridges with the presence of particles in the contact zone itself, the fluid film may help to displace the particles to expose the adhesive tips. The thin fluid film may behave like a lubricant [37], facilitating the movement of accumulated particles. The low-resistance sliding of small particles as they are shed can be seen in the electronic supplementary material, video SV2. A similar effect was also found in the self-cleaning of adhesive pads in tree frogs [12] where beads particularly accumulated in the deposited fluid left behind as ‘footprints’. When fluid is absent, like in many synthetic fibrillar adhesives [10], self-cleaning of small particles is drastically reduced. Adhesive pads without a fluid, like those in geckos and spiders, tend to have dense, tightly packed fibre arrays, which may help prevent lodging of microparticles [5,38]. Interestingly, in simulated steps of restrained beetles [3], adhesive forces recovered much faster. It is unclear, though, whether beetles in this situation would produce similar amounts of fluid to the natural condition.

Our study indicated a relationship between self-cleaning performance and particle size consistent with findings of previous studies [3,4,10,11]. In electronic supplementary material, figure S1 and table S2, we compare our findings for unrestrained beetles to those previously reported for restrained beetles [3]. The self-cleaning rates for small particles in unrestrained and restrained beetles are almost identical, indicating that the restrained condition mimicked...
the natural one well enough (e.g. length of slippage) to show a similar cleaning. However, the self-cleaning rates for medium and large particles differ.

For large particles, we observed unrestrained beetles removing more particles than restrained beetles. Therefore, unrestrained gait kinematics do play an important role in self-cleaning of large particles. In high-speed video observations (for example, see the electronic supplementary material, video SV2), we observed beetles removing large particles that were attached to the sides of their adhesive pads (as shown in figure 2a–g). These particles can only be removed through unrestrained motions of the footpads, especially by tilting the pads so particles can contact the substrate. However, the particles removed during later steps may not be crucial for adhesion, as the previous study found that the restrained beetles fully recovered adhesion after three steps [3].

For medium particles, a previous study reported that only the first step removed particles for restrained beetles [3], while we observed particles being removed throughout all 10 steps. This difference is made apparent through the decay rates of the exponential fits shown by the solid lines in figure 3a–c. As shown in table 1, the removal of beads per step decays slowest for the medium beads, while the fastest decays were observed for the small beads. On the other hand, restrained beetles showed the fastest decay rates for the medium beads (electronic supplementary material, table S2). The simulated steps in the previous study by Clemente et al. [3] used a 0.5-mm long shearing drag only, whereas we observed a much larger variation in slip lengths in climbing beetles (0.05–4.5 mm, with a median value of 0.66 mm). Presumably, as the hairs get bent by the proximal drag, beads will move through the fibres and eventually either reach the distal end of the pad or come closer to the surface where they can adhere to the substrate. A medium particle being deposited onto the substrate is shown in the electronic supplementary material, video SV2. This process presumably takes longer as the particles are lodged in between the hairs and are less likely to transfer immediately to the substrate. Furthermore, we observed that particles get trapped underneath the adhesive tips of the setae (figure 2e), and thus would require even longer to be removed through surface contact. For these particles, grooming may be required. While beetles lack specialized appendages for grooming, like those found on ants [39] and bees [40], they have been observed to use their mandibles to aid in grooming [4]. Grooming is less desirable than self-cleaning, but it has been found to be an effective technique for cleaning in insects [40,41].

Our study focused exclusively on the male green dock beetle, which is a model organism for studying biological fibrillar adhesives [3,13–17]. However, future work comparing self-cleaning rates between other insect species could be fruitful. Specifically, such work could help further address the role of setal morphology on self-cleaning. The seta length, thickness and inter-setal spacing can vary significantly between species, especially across orders, like Diptera (flies) and Coleoptera (beetles) [38].

While our study is not the first to identify self-cleaning mechanisms for biological adhesives, we presented those implemented by green dock beetles for cleaning micrometre-scale, fibrillar adhesives. Self-cleaning in these adhesives may vary from the self-cleaning exhibited by the nano-scaled adhesives of geckos, which a recent study found to be highly dependent on the pull-off velocity [8]. The pull-off velocity dictated crack propagation within the
contact zone. Because our contaminants were of a similar scale to the adhesive fibres, we observed interlocking that was only overcome through shearing and rolling of the particles. However, crack propagation and contact zone mechanics may still play a role. Our study did not investigate pull-off velocity, but this may be of interest for future work.

4.2. Effect of surface energy and roughness of the substrate on self-cleaning

In addition to particle size, we found that the substrate plays a critical role in particle accumulation and removal. Using atomic force microscopy, we measured the adhesive force required to detach small, medium and large beads from a glass surface, a leaf surface and the adhesive pad. Overall, the beads had a lower affinity to the flat glass surface than to the pad, which allowed the pad to pick up beads and become soiled. This was different for the leaf surface as we found an overall higher affinity of the beads to the surface compared with the affinity to the pad. This could explain why so few particles were picked up (less than 100 of medium beads and less than 20 of large beads).

In our climbing trials on a dock leaf, we found also that much fewer particles were deposited. Shown in figure 7b–d, the leaf gives rise to surface features with heights up to 25 μm and widths of roughly 50 μm from veins, cell wall connections and stomates (pores). As claws may interlock on such rough substrate [14], the slip lengths were significantly reduced with consequently less particles deposited. However, our force measurement also showed that the particles had a higher affinity to the leaf and thus pads have picked up less particles to begin with (60 and 16 beads on the leaf substrate versus 1900 and 180 beads on the glass surface for medium and large beads, respectively).

Interestingly, dock leaves exhibit a more hydrophilic surface (water contact angle of 49°) compared to the hydrophobic nature of most other leaves covered by a waxy layer [42]. Previous studies have found the dock leaf to be one of the ‘stickiest’ plants, having a low contact angle and high adhesion [43,44]. The surface free energy of the leaves also has a higher contribution from dispersive rather than polar surface free energy, as shown in electronic supplementary material, table S1. The surface free energy of polystyrene (the material of the microbeads used) is all dispersive [33], which may explain the high affinity of beads to the leaf. Previous studies have demonstrated that the adhesive fluids of insects with smooth pads exhibit stronger adhesion when the dispersive contribution is higher and the polar contribution is lower [15]. Additionally, previous studies also found that adhesion in dock beetles is strongly influenced by hydrophobicity [17]. Future studies quantifying the surface free energy (dispersive and polar) of the adhesive fluids of insects, as well as pollen and other contaminants, may show interesting aspects regarding self-cleaning of natural contaminants.

Our study focused on hydrophilic surfaces since dock beetles live on hydrophilic leaves. A previous study by Orchard and colleagues investigated the recovery of climbing insects

### Table 3. Statistical comparisons of adhesion according to substrate. All comparisons were conducted using the Kruskal–Wallis test. p-Values were adjusted for multiple comparisons using the Bonferroni correction.

<table>
<thead>
<tr>
<th>comparison (μm)</th>
<th>substrate</th>
<th>d.f.</th>
<th>$\chi^2$</th>
<th>p-value</th>
<th>N</th>
</tr>
</thead>
<tbody>
<tr>
<td>1 versus 10</td>
<td>glass</td>
<td>(1, 8)</td>
<td>0.0667</td>
<td>&gt;0.05</td>
<td>3</td>
</tr>
<tr>
<td>1 versus 10</td>
<td>leaf</td>
<td>(1, 9)</td>
<td>0.1169</td>
<td>&gt;0.05</td>
<td>3</td>
</tr>
<tr>
<td>1 versus 10</td>
<td>adhesive pad</td>
<td>(1, 14)</td>
<td>1.339</td>
<td>&gt;0.05</td>
<td>7</td>
</tr>
<tr>
<td>1 versus 45</td>
<td>glass</td>
<td>(1, 12)</td>
<td>0.1143</td>
<td>&gt;0.05</td>
<td>3</td>
</tr>
<tr>
<td>1 versus 45</td>
<td>leaf</td>
<td>(1, 11)</td>
<td>2.470</td>
<td>&gt;0.05</td>
<td>3</td>
</tr>
<tr>
<td>1 versus 45</td>
<td>adhesive pad</td>
<td>(1, 14)</td>
<td>4.339</td>
<td>0.07</td>
<td>7</td>
</tr>
<tr>
<td>10 versus 45</td>
<td>glass</td>
<td>(1, 15)</td>
<td>0.1059</td>
<td>&gt;0.05</td>
<td>6</td>
</tr>
<tr>
<td>10 versus 45</td>
<td>leaf</td>
<td>(1, 15)</td>
<td>1.751</td>
<td>&gt;0.05</td>
<td>7</td>
</tr>
<tr>
<td>10 versus 45</td>
<td>adhesive pad</td>
<td>(1, 15)</td>
<td>6.353</td>
<td>&lt;0.05</td>
<td>8</td>
</tr>
</tbody>
</table>

### Table 4. Statistical comparisons of adhesion according to bead size. All comparisons were conducted using the Kruskal–Wallis test. p-Values were adjusted for multiple comparisons using the Bonferroni correction.

<table>
<thead>
<tr>
<th>comparison</th>
<th>bead size (μm)</th>
<th>d.f.</th>
<th>$\chi^2$</th>
<th>p-value</th>
<th>N</th>
</tr>
</thead>
<tbody>
<tr>
<td>adhesive pad versus glass</td>
<td>1</td>
<td>(1, 9)</td>
<td>3.753</td>
<td>0.11</td>
<td>3</td>
</tr>
<tr>
<td>adhesive pad versus leaf</td>
<td>1</td>
<td>(1, 9)</td>
<td>1.052</td>
<td>&gt;0.05</td>
<td>3</td>
</tr>
<tr>
<td>adhesive pad versus glass</td>
<td>10</td>
<td>(1, 13)</td>
<td>5.400</td>
<td>&lt;0.05</td>
<td>6</td>
</tr>
<tr>
<td>adhesive pad versus leaf</td>
<td>10</td>
<td>(1, 14)</td>
<td>1.340</td>
<td>&gt;0.05</td>
<td>7</td>
</tr>
<tr>
<td>adhesive pad versus glass</td>
<td>45</td>
<td>(1, 17)</td>
<td>0.000</td>
<td>&gt;0.05</td>
<td>6</td>
</tr>
<tr>
<td>adhesive pad versus leaf</td>
<td>45</td>
<td>(1, 16)</td>
<td>5.787</td>
<td>&lt;0.05</td>
<td>7</td>
</tr>
</tbody>
</table>
on hydrophobic and hydrophilic surfaces [4]. In their study, they found that surface free energy had a significant effect on the recovery of adhesion for insects with hairy footpads. Overall, the beetles took longer to recover on hydrophobic surfaces. This insight may explain why the dock beetles thrive on the hydrophilic leaves of the dock plant.

4.3. Translation into engineering

There have been great strides towards realizing bio-inspired, reversible, dry adhesives [18–26], but only a few studies have addressed the issue of self-cleaning [8,10,11]. Self-cleaning of particles of a certain size has been identified as a detrimental issue for fibre-based synthetic adhesives [10,11]. These systems, just like their biological counterparts [3], exhibit lodging of particles that fit between their fibres, which, in turn, have negative effects on adhesion.

This study presents possible ideas to address the issue of self-cleaning lodged particles. In order to shed lodged particles, beetles used cleaning stops where the adhesive pads were dragged for long distances and then quickly returned for another dragging step. We found that both the number of particles removed and slip length decreased significantly as the beetles climbed, revealing a correlation between slipping and cleaning. Previous studies have also demonstrated the importance of slipping (or shearing) in cleaning [3,10]. While the slips may be involuntary when adhesion fails, their implementation led to the eventual recovery of adhesion. Slipping thus provides a passive mechanism that may be implemented in climbing robots or grippers to promote self-cleaning without additional programming or specialized structures. Besides flushing adhesives with water or specially designed grooves for collecting sheared particles [10,45], there are no strategies currently employed for synthetic systems to cope with soiling.

Additionally, the use of adhesive fluids in concert with fibre structures may help improve adhesion when soiled with small particles, as mentioned earlier. While the residue left by adhesive fluids may not be ideal for certain applications, they may be employed for dealing with situations where soiling from micro-particles is unavoidable.

Like previous studies, a key geometrical relationship we found to dictate cleaning performance was the ratio between particle size and inter-setal distance. This ratio determined particle lodging and penetration. However, another geometrical relationship of importance may be the ratio between particle size and fibre length [10]. This parameter also dictates particle penetration. The diameter of the large particles in our study was roughly the same as the length of the beetle’s adhesive fibres, which indicates that the large particles were always able to make contact with the substrate. Constant contact with the substrate promotes rolling and shearing, and may contribute to their effective self-cleaning.

5. Conclusion

This study quantified the contact self-cleaning performance of fibrillar adhesive pads on freely climbing dock beetles. Compared to restrained beetles, freely climbing beetles demonstrated a robustness to soiling from particles of small (1 μm), medium (10 μm) and large (45 μm) sizes. Gait kinematics were significantly affected by soiling, with soiled pads slipping 10 times further than clean pads. While pad slipping may negatively affect climbing performance, we found that longer slip lengths removed more particles. Additionally, soiled pads spent more time in contact with the substrate than clean pads. In short, beetles were found to employ dedicated, and possibly involuntary, cleaning steps where the adhesive pads were dragged for long distances and then quickly returned for another dragging step. Such strategies could be adapted to synthetic adhesive systems, like climbing robots or grippers. Overall, we found that particle size and climbing substrate play a crucial role in dictating cleaning and climbing performance. Small particles were the hardest to remove but affected climbing performance the least. Medium particles had the most detrimental effects on climbing performance, in partial agreement with the previously reported fixed footpad cleaning experiments [3]. Large particles were cleaned most effectively. Adhesion of particles to smooth, stiff glass was lower than on rough, soft dock leaves, so soiling and cleaning was found to be lower for leaves. Overall, pad slippage and high adhesion of dock leaves were found to promote effective self-cleaning and reduced fouling.

Data accessibility. The datasets supporting this article have been uploaded as part of the electronic supplementary material.

Author’s contributions. G.J.A. designed the study, carried out the experiments, participated in data analysis and drafted the manuscript; T.E. carried out the data analyses, participated in the experiments, participated in the design of the study and drafted the manuscript; M.S. participated in the design of the study and drafted the manuscript. All authors gave final approval for publication.

Competing interests. We declare we have no competing interests.

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References
