

Bacterial Photonic Pigments

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Abstract

The brightest colourations in nature are produced by nanostructured materials at the scale of visible light wavelengths. Living organisms make these photonic architectures from biomaterials alone under ambient conditions, motivating their use as sustainable alternatives to synthetic pigments. However, no scalable route has yet been developed to convert living photonic organisms into processable pigment powders. Here, we demonstrate that naturally occurring structurally coloured bacterial colonies can be directly harvested to produce structurally coloured pigments using scalable high-throughput methods. Specifically, we developed a continuous growth system in which structurally coloured biofilms grown on membranes can be harvested, fixed, and finally pulverised into pigments ready for integration into formulations. Red, green, and blue pigments spanning the visible spectrum are achieved by varying growth temperature, applying chemical post-treatments, using genetically modified mutants, or employing sol-gel methods. These results demonstrate that the self-organisation of living cells can replace synthetic chemical fabrication routes, yielding structural colour pigments composed primarily of biomaterials.

Introduction

Whilst the generation of pigments and dyes relies on the selective absorption of light, structural colouration arises differently, from constructive interference of light by periodic nanostructures¹. In multicellular, eukaryotic organisms, structural colour is produced with a wide range of architectures and materials, ranging from the periodic packing of collagen fibre in mammals² to the complicated 3D complex chitin-containing composite nanostructures observed in insects^{3–5}, to guanine nanoplatelets and nanoparticles in sea creatures^{6,7} and reptiles⁸, to cellulose and wax architectures in plants.^{9–11}

While such bright, iridescent colours are widespread in the natural world¹², they are increasingly present in our everyday lives¹³. In fact, inspired by nature, a wide variety of complex nanoscale architectures have been proposed to produce structural colour materials and pigments using different fabrication routes,^{14,15} from top-down techniques^{16,17} to bottom-up processes such as the self-assembly of colloidal particles.^{18–21,22,23}

Despite the significant progress and success of the aforementioned approaches, most require chemical synthesis or isolation of nanoscale building blocks. A route in which the building blocks self-grow, self-replicate, and self-organise into photonic order without external fabrication offers a significant sustainability advantage.

Here, we therefore propose to use bacterial cells themselves as nano-building blocks to produce photonic structures. Certain rod-shaped bacteria provide exactly this opportunity. Several strains can, in fact, self-arrange into a well-ordered two-dimensional photonic crystal, resulting in strong and directional colouration.^{24–29} Previous works have shown that the colour, which depends on the spatial arrangement

of the bacteria,^{26,27} can be tuned either naturally by changing environmental growth parameters^{30,31} or engineered via genetic manipulation^{26,32} and even patterned on different substrates³³.

Moreover, when combined with the potential to grow them cheaply on a large scale without chemical processes, bacteria appear to be a ideal candidate for producing vibrant, non-toxic, structurally coloured pigments, representing a cheap and sustainable alternative to traditional colloidal building blocks.

Here, we demonstrate a scalable, agar-free method to grow bacterial-based photonic pigments. Specifically, we grow structurally coloured bacterial biofilms at the air-liquid interface on a semipermeable membrane. The use of membranes simplifies the growth, control and harvest of intact, large-area biofilms without compromising their optical performance, while membrane recycling simultaneously reduces the amount of medium required and the waste generated. Once harvested, the coloured biofilms can be directly post-processed to produce pigments, providing an intrinsic path to continuous, scaled-up production.

Moreover, our flow-in membrane system allows in situ post-processing of the biofilms using different fixation procedures, including both crosslinkers and sol-gel techniques, the latter leading to fully inorganic inverse-opal-like pigments.

Finally, we demonstrate that red, green, and blue pigments can be obtained by altering growth conditions or through post-fixation chemical treatments, establishing a scalable, sustainable workflow to produce structurally coloured, free-standing bacterial biofilms with improved mechanical stability and simplified handling.

As microbial colonies are known to exhibit emergent, hierarchical internal organisations, and sol-gel chemistry offers a vast range of materials that decouples resulting material properties from biological templates, our results establish living, adaptive microbial architectures as a broad platform for diverse functional nanomaterials.

Results

Free-standing structural coloured bacterial pigment flakes

To demonstrate our method for fabricating large, free-standing, structurally coloured bacterial films, we selected the strain *Cellulophaga lytica* PLYA2³⁴, and *Flavobacterium IR1* and its mutant M16,²⁶ and we believe that the results can be generalised to other strains capable of producing structural colours.

The fabrication concept used to develop bacterial pigments is presented in **Figure 1a** and consists of a few simple steps: first, the inoculation and incubation of the biofilm onto a porous membrane mounted on a continuous-flow liquid chamber; then, an *in-situ* fixation by substituting the growth medium with a crosslinking solution diffusing through the membrane; lastly, the detachment of the fixed biofilm as a free-standing photonic film and its pulverisation to achieve structurally coloured pigments.

The agar-free culturing device with instantaneous physiochemical control is shown in **Figure 1b**, which consists of a liquid reservoir with inlet and outlet ports, sealed on its upper surface by a semi-permeable membrane. In this configuration, the membrane facilitates organisation and serves as a physical barrier between the biofilm and the liquid medium, while allowing bidirectional material transport and facilitating bacteria's self-organisation. During colony growth, fresh nutrients diffuse upward through the membrane to the cells, while metabolic waste products are carried away by convective flow beneath the chamber. A liquid medium can be pumped through the chamber at a slow rate (1 mL/hr), maintaining a moist, flat membrane surface that supports uniform biofilm formation across the entire membrane area. This represents a substantial advantage over agar-based cultivation: most nutrients in the agar substrate are left unconsumed during the growth cycle, and the brittle agar substrate impedes integration with continuous processing, automation, or recycling. This single limitation cascades into every practical barrier from the lab to large-scale production, concerning cost, material conversion, and waste, which has prevented the translation of microbial-based living materials into functional materials.

We tested several membrane materials, including dialysis tubing (regenerated cellulose), cellophane sheets, and laboratory-grown bacterial cellulose. All tested membranes supported biofilm growth and yielded comparable optical performance (**Table S1**, Supporting Information). The membrane selection spans a range of practical considerations: dialysis tubing offers a well-defined molecular weight cut-off for controlled transport; cellophane is inexpensive and industrially available; and bacterial cellulose is entirely bio-derived, opening a route to an all-biological production chain. Importantly, this approach is not constrained to a specific substrate chemistry. Any membranes that are biologically compatible and have sufficient porosity can be used, provided they create physical separation while allowing material transport. Unless otherwise specified, results reported below were obtained using dialysis tubing membranes as representative substrates.

After the device is filled with liquid medium, bacteria are inoculated onto the membrane, and the device is kept at room temperature (25 °C). A brightly coloured biofilm with a thickness of a few tens of microns can be observed after 12 hours of incubation (**Figure 1b**). At this point, the biofilm can be directly fixed by introducing an aldehyde-based fixative solution (see Methods section) through the device inlet, thereby replacing the growth medium in the chamber. After injection, the fixative rapidly diffuses through the porous membrane, reaches the bacterial biofilm, and crosslinks it *in situ*, increasing its mechanical strength sufficiently to be harvested simply by detaching it from the membrane. This process therefore yields a large, robust, and freestanding film (**Figure 1c**) whose size depends only on the membrane dimensions, and can subsequently be ground into flakes readily used as photonic pigments (**Figure 1d**).

Several features of this workflow distinguish it from conventional cultivation in terms of scalability. First, it allows post-processing of the bacterial-only free-standing film into a pigment powder. Second, the membrane can be sterilised and reused after each growth cycle, eliminating single-use substrate waste and substantially reducing material costs. A rough, materials-only estimate (**Table S2**, Supporting Information) indicates that, per unit area of biofilm, the conventional agar substrate is approximately

fivefold more costly than the reusable cellophane membrane at laboratory prices and roughly twentyfold more costly in bulk. Moreover, the membrane area, hence the pigment yield, has no intrinsic upper bound. Membranes can be fabricated at arbitrarily large dimensions or geometries and arranged compactly in three dimensions to multiply throughput. Neither of these is feasible with agar, which is consumed after each cycle and does not support vertical integration. Third, because medium exchange, fixation, and rinsing are performed by pumping solutions through the chamber, the entire process from growth to harvest can be automated with minimal manual intervention, allowing independent control over nutrient composition, flow rate, and fixation timing. Together, these attributes establish a fabrication pipeline that is amenable to continuous, scaled-up production.

Expanding the colour palette

As in any other colloidal-based photonic pigment, the reflected colouration depends on the periodic organisation and the size and refractive index of the scattering elements, as well as the matrix that contains them²⁷. In the case of bacterial structural colouration, the scattering centres are the rod-shaped cells, which are periodically arranged in a two-dimensional photonic crystal with a hexagonal geometry²⁷. The colour of a bacterial colony can be changed by genetically engineering the strain²⁶, altering the incubation conditions,³⁰ or post-treating the fixed biofilm. Here, we use a combination of these methods to obtain free-standing bacterial photonics films across the visible spectrum (See **Figure S3**, Supporting Information), which we subsequently grind to produce pigments (**Figure 2a-i**). Smaller photonic pigments can be obtained by further grinding or ball milling (an example of a differently sized pigment from agar biofilms is shown in **Figure S4**, Supporting Information).

In the case of *C. lytica* PLYA2, biofilms grown under standard conditions exhibit a strong green colouration (**Figure 1b**), and, once fixed with glutaraldehyde and formaldehyde (see Methods), these free-standing films can directly yield green pigments (**Figure 2d-f**). To achieve blue pigments, the fixed green biofilms were post-treated with butyl glycol, which dehydrates the biofilm, inducing a contraction of the photonic lattice and shifting the reflectance peak to 420–450 nm (**Figure 2g-i**). To achieve red-coloured biofilm and pigments, we instead used *Flavobacterium* IR1-M16, a mutant of IR1 that naturally displays a red colouration with well-defined reflectance peak spanning 625-645 nm.

The morphological features and the overall organisation of the bacterial pigments were characterised via scanning electron microscopy (SEM). The representative SEM images shown in **Figure 3** and **Figures S5** and **S6** (Supporting Information), indicate that the fixation and fragmentation processes preserve the overall thickness of the biofilm (tens of microns, **Figure 3b**) and the periodic organisation of the cells (**Figure 3c**), ensuring that the structural colour is well retained. Similarly, angle-resolved spectroscopy confirms the preservation of the photonic structure, as shown in **Figures S7** and **S8**, which are discussed in detail in the Supporting Information.

Transforming bacterial architectures to inorganic materials: bacterial colonies as a template for inverse-silica architectures

The free-standing films presented above carry the photonic structure in fixed organic form, tying the material's properties to the bacteria that built it. Decoupling the architecture and material properties enables a far-reaching possibility that utilises both the complex, emerging internal structures observed in bacterial colonies and the vast material choices afforded by synthetic chemistry. On the structural side, the 2D bacterial photonic crystal demonstrated above represents one example of a much larger structural repertoire: microorganisms are known to generate a remarkable diversity of ordered architectures through collective self-organisation, from vortex lattices, active turbulence, to hierarchical three-dimensional biofilm networks, many of which have no synthetic equivalent and remain inaccessible to either top-down or bottom-up fabrication. Therefore, we decided to use sol-gel chemistry as a natural entry point, in which infiltration of a template followed by calcination to remove the organics can create a broad family of metal oxides, including silica, titania, zirconia, and their hybrids, each offering different refractive indices, bandgap properties, and chemical functionalities.

To demonstrate this concept, we utilised the bacteria biofilm on the membrane as a living template for sol-gel chemistry with a silica precursor to obtain inverse inorganic photonic materials similar to previously fabricated inverse opals and cholesteric structures.^{35,36} this

Specifically, we first infiltrated a structurally coloured *C. lytica* PLYA2 biofilm with silica sol, derived from pre-hydrolysed tetraethyl orthosilicate (TEOS) under acidic conditions. The sol suspension was introduced through the inlet of the membrane chamber, repurposing the same flow architecture used for growth and fixation. As water evaporates from the upper surface of the biofilm, convective transport draws the sol particles (of a few nm in diameter)^{37,38} upward through the porous membrane and into the nanostructured biofilm. The silica particles accumulate progressively within the biofilm matrix, filling the interstitial spaces between the bacterial cells, similar to cake filtration in a standard separation process. The system was left overnight to ensure complete infiltration, after which the composite film was removed from the device and dried for 2 hours under ambient conditions.

The resulting silica–biofilm composite preserves both the structural integrity and the photonic response of the original colony. SEM cross-sections reveal the bacterial cells embedded within a continuous silica gel matrix (**Figure 4a,b**), which provides sufficient mechanical support to prevent cell collapse during drying. Optical microscopy confirms that the structural colour is largely unchanged at this stage (**Figure 4c,d; Figure S9**, Supporting Information), consistent with the low refractive index contrast between the silica matrix and the organic extracellular material of the biofilm.

The composite was then subjected to a two-stage thermal treatment, in which the sample was heated at 200 °C for one hour to consolidate the amorphous silica network, followed by calcination at 600 °C (ramp rate 2 °C min⁻¹) to remove all organic material and fully densify the silica matrix. After calcination and slow cooling to room temperature, the organic template is eliminated, leaving behind a silica inverse

photonic structure in which the periodic array of voids corresponds to the positions of the original bacterial cells (**Figure 4e,f**). The photonic architecture of the biological template is thus transformed and preserved in the inorganic form. The optical response is blue-shifted and broadened relative to the pre-calcination composite (**Figure 4g,h**), reflecting the increased refractive-index contrast between silica and air, as well as densification-induced shrinkage.

However, it is important to note that in this method, the membrane cannot be removed prior to calcination (**Figure S10**, Supplementary Information), leading to additional scattering of the optical response.

Conclusions

In conclusion, we demonstrate a scalable methodology for producing photonic pigments using bacteria as building blocks. Specifically, we harness the ability of certain rod-shaped bacteria to self-organise during growth into photonic crystals, then fix and pulverise them into microparticles without loss of colour intensity, yielding pigment flakes with colours across the visible spectrum. The ease of fabrication and the ability to configure the system for continuous operation using recyclable semi-permeable membranes indicate the potential to scale up to larger surfaces for higher yields of bacterial pigments

These capabilities suggest a different basis for the fabrication of structurally coloured materials. In conventional routes, the optical material and the ordered architecture are coupled. The material is typically synthesised in the form of (near) monodisperse particles, which are then suspended and assembled into a periodic structure either by evaporation or by gravity-induced crystallisation processes. The bacterial system bypasses these requirements, as the building blocks and the architecture are grown rather than synthesised, and self-organised rather than externally assembled. Importantly, the material can be independently selected via the sol-gel method, which offers a broad design space for engineering optical properties.

While the platform operates at laboratory scale, it has the potential for larger-scale production as it is based on a continuous-flow architecture, recyclable membranes, and compatibility with automation. Beyond pigments, the templating route points towards inorganic photonic materials that combine architectures difficult to achieve by synthetic assembly with material chemistries inaccessible to biology. Replacing silica with a higher-refractive-index oxide such as titania would increase the dielectric contrast towards a photonic bandgap, extending the approach from colourants to light management and sensing. More broadly, microorganisms self-organise into a far wider range of ordered structures than the hexagonal lattice templated here, from dynamically assembled vortex arrays to three-dimensional biofilm networks, many of which have no synthetic counterpart. As such, sacrificial templating of such microbial structures offers a route to ordered inorganic architectures that remain inaccessible to current top-down and bottom-up fabrication.

Methods

Bacterial culture, biofilm growth, and chemical treatments.

The bacterial strains used in this study are *Cellulophaga lytica* strain PLYA2, isolated from Plymouth (UK) and *Flavobacterium* IR1 mutant M16, both provided by Hoekmine BV.

Agar-free growth chambers. The custom setup was designed to support a flat, hydrophilic membrane that enables continuous nutrient exchange between the liquid media and the bacteria. Different types of membranes were used: dialysis tubing membranes from regenerated cellulose (MWCO: 14,000 Da, membra-cel™, Carl Roth), Gel drying frames made of cellophane (MWCO~12,000 Da, Merck), and bleached bacterial cellulose (lab-grown, see below). Before inoculation, the setup was sterilised with 70% (v/v) ethanol, followed by UV exposure. The membranes were hydrated with sterile liquid growth medium and mounted under tension. The inoculation was performed with an L-shaped spreader. The growth medium consists of Peptone (3.75 g/L, Sigma-Aldrich), Yeast extract (1.25 g/L, Sigma-Aldrich), sea salt (32.5 g/L, Lima), and magnesium sulfate (375 mg/L, Fisher Scientific) dissolved in milliQ water. Nigrosine (0.15 g/L, Sigma-Aldrich) was always added to improve the visual contrast. The biofilms were incubated at 25°C for one day. Bacterial cellulose membranes were grown using *Komagataeibacter hansenii*, cultured for 5 days in HS liquid medium in square Petri dishes (245 × 245 × 25 mm³). Once a cellulose pellicle approximately 500 µm thick had formed, the bacterial cellulose was harvested and purified by boiling for 20 min, followed by two consecutive NaOH treatments (10 min each), and then autoclaved to remove residual bacterial cells.

Growth on agar plates. For growth on agar plates, the same media recipe was used, plus the addition of Agar (12.5 g/L, Invitrogen). After inoculation, plates were left to dry for 10 minutes and then incubated at 20 °C for 1 day, after which they reached optimal photonic response. Biofilms grown onto agar plates were used to study the butyl glycol-induced blue-shifting.

Chemical blue-shifting. To obtain blue hues, fixed biofilms of *C. lytica* PLYA2 were covered with 6 mL of butyl glycol (Sigma-Aldrich) overnight and subsequently rinsed twice with 3% saline solution.

Fixation and biofilm detachment. To preserve the two-dimensional photonic structure, biofilms were fixed with an aqueous fixative. (1) In the case of the biofilms growing on agar plates, 6 mL of a fixative solution containing 1% v/v formaldehyde, 6% v/v glutaraldehyde, and 50% v/v ethanol were poured on top and left for 5 minutes, and subsequently rinsed twice with a solution of 50% v/v ethanol, 5% v/v glycerol for 5 minutes each time. After fixation, the biofilm can be detached from the agar substrate by gently pipetting the aqueous solution between the biofilm and the agar. These free-standing, floating biofilms (or their fragments) were then collected and stored in an aqueous solution containing 50% v/v ethanol and 5% v/v glycerol, in which they remain stable for several years. (2) The biofilms grown on permeable membranes were fixed via diffusion of fixative containing 2% v/v formaldehyde, 2% v/v glutaraldehyde, in 3% w/v saline solution into the chamber for 25 min. The biofilms were then rinsed twice with a solution of 30% v/v ethanol and 5% v/v glycerol to remove any residual fixative. The biofilms were detached by

submerging the membrane in a 3% w/v saline solution, then stored in an aqueous solution containing 30% v/v ethanol and 5% v/v glycerol. The membranes were thoroughly rinsed to remove any residual material and reused for the next growth cycle.

Fixation via Silica nanoparticles: Tetraethyl orthosilicate (TEOS), 0.1 M hydrochloric acid (HCL), and ethanol in a volumetric ratio 1:1:1.5 were stirred at room temperature for one hour to pre-hydrolyse the TEOS. The biofilm was then infiltrated with a dilution of pre-hydrolysed TEOS in Milli-Q water at a 1:3 (v/v) ratio, allowing the solution to diffuse for at least 5 hours.

Biofilm Calcination: After TEOS diffusion and drying, the resulting membranes underwent two thermal treatments. Samples were first heated at 200 °C for 1 h to promote further condensation and consolidation of the amorphous silica network; this was followed by calcination at 600 °C with a ramp rate of 2 °C min⁻¹ to remove organic components and fully densify the silica matrix, and were then left to cool overnight.

Fabrication of microparticles. To fabricate micron-scale pigment flakes, the fixed, free-standing biofilm can be broken into smaller flakes using a vortex mixer or with ball-milling. The solvent formulation used for both the harvesting process and long-term storage is 30% v/v ethanol and 5% v/v glycerol in water. This composition keeps the structure hydrated and prevents the growth of unwanted microorganisms.

Microscopy and spectroscopy.

Digital microscopy, optical microscopy and micro-spectroscopy. To characterise the colour, optical imaging and micro-spectroscopy were performed using a customised Zeiss AxioScope 7 equipped with a 20x objective (Zeiss, Apochromat, NA 0.6) and using a halogen lamp as light source (Zeiss, HAL100). For the measurement, the pigment flakes were kept in an appropriate solvent mixture on a glass slide by using imaging spacers (Grace Bio-Labs SecureSeals, 120 µm depth) and covered with a coverslip. Reflectance spectra were recorded on an area of 20 µm in diameter by coupling the microscope to a spectrometer (Avantes, AvaSpecHS2048) via an optical fibre (Avantes, FC-UVIR100-2-SR, 100 µm core size) and normalised against a white standard (Spectralon). Micrographs were acquired with a CMOS camera (Pixelink, PL-D725CU-T, calibrated against a white standard). Images and reflectance spectra were acquired in dark field mode, unless otherwise stated. A Keyence digital microscope (VHX-7000) equipped with E20, E100, and E500 lenses was used to obtain low magnification images. Photographs were taken with a digital camera (Nikon D7500) at a fixed working distance and illumination, calibrated against a white background.

Scanning electron microscopy. To obtain the microparticles in a dry state while preserving their structure, the samples were first fully dehydrated with an alcohol series (ethanol 50% v/v, 70%, 80%, 90% for 10 mins each, then 100% 3 times for 30 mins each) and then dried with CO₂ via critical point drying (Leica, EM CPD300). The dried pigments were then stored in vials. To characterise their morphology, the free-standing microparticles were mounted on aluminium SEM stubs with carbon tape and coated with 10

nm Pt using a sputter coater (Quorum, Q105T ES), and subsequently observed on a TESCAN Mira3 at 3kV acceleration voltage and a working distance of 5-6 mm.

Optical goniometry. To characterise the photonic crystal structure of the bacterial biofilm, reflectance spectra were recorded at different angles of incidence and detection using an in-house-built angular-resolved spectroscopy system. For biofilms grown on agar, a 1 cm² square of film was cut from the plate and mounted on a glass slide. Free-standing biofilms are instead kept in a solvent mixture on a glass slide by using imaging spacers (Grace Bio-Labs SecureSeals, 120 µm depth) and covered with a coverslip. The sample was illuminated on an area of ~2 mm diameter with a Xenon lamp (OceanOptics, HPX2000) connected to a collimator (Thorlabs, RC08SMA-F01) via an optical fibre (Avantes, FC-UVIR100-2-SR, 100 µm core size). The reflected light was collected from the same area using another collimator (Thorlabs, RC08SMA-F01) connected to a spectrometer (Avantes, AvaSpec-HS2048) via an optical fibre (Avantes, FC-UVIR100-2-SR, 200 µm core size). Reflectance spectra were normalised measuring a white standard (Spectralon) with 0° incidence and 5° collection.

Declarations

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Author contribution statements

SV and CI conceptually developed the idea of the development of photonic pigments based on bacteria, JW and JG developed the membrane system, MM performed all the measurements to lead the EM and optical characterisation. YO and RP helped with the photonic pigments development, RH, LC, CI, and MP developed the strain and helped with cultivation. PB was involved in post-fixation colour optimisation. SV and MM prepared the draft, and all the authors contributed to the editing.

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Figures

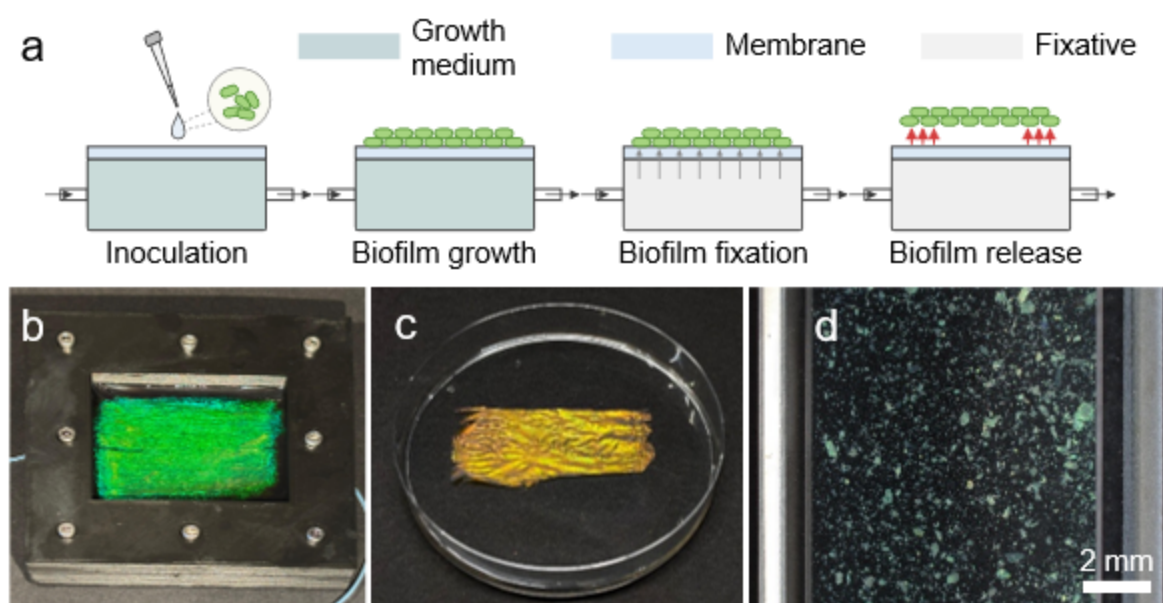


Figure 1

Fabrication of free-standing bacterial pigment flakes. Agar-free biofilm fabrication setup. a) Schematic of the continuous flow fabrication system that allows for bacterial growth, media exchange and fixation. Black arrows indicate the flow of the liquid. Bacteria are inoculated from liquid culture onto a porous membrane placed above a liquid medium in a custom 3D-printed chamber measuring 5 cm × 6 cm × 0.5 cm (L × W × H). After a 24 h incubation period, the medium is replaced with fixative, which diffuses upward through the membrane. After a 25-minute exposure, the biofilm can be detached from the membrane and ground into bacterial flakes. b) Photography of a living biofilm grown on the fabrication system (as in 2). c) After fixation, the biofilm gains sufficient mechanical stability to be completely detached from the membrane. d) Digital micrograph of bacterial flakes in an aqueous solution (50% v/v ethanol, 5 % v/v glycerol).

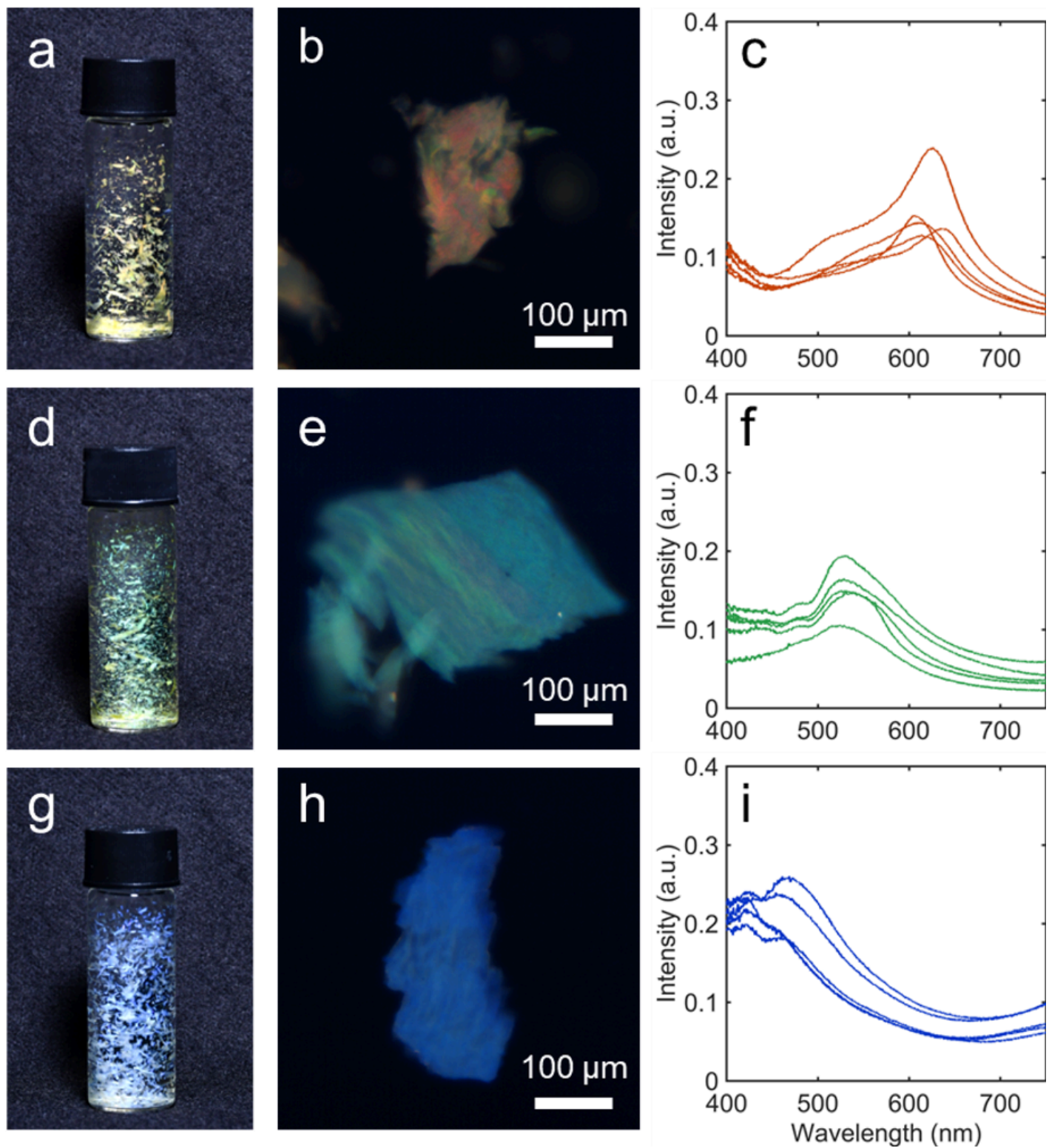


Figure 2

Optical appearance of bacterial pigment flakes. Optical appearance of red (top row), green (centre), and blue pigment flakes (bottom). a, d, g) macroscopic appearance in glass vials with a diameter of 1 cm. f; b, e, h) representative micrographs; c, f, i) reflectance spectra of five different flakes.

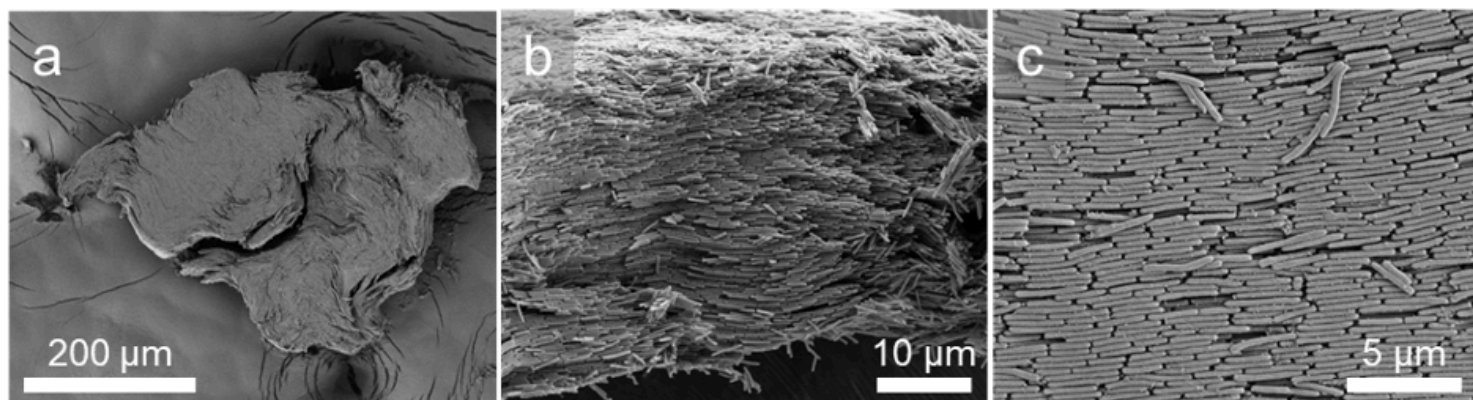


Figure 3

Morphological characterisation of bacterial flakes. Morphology of bacterial large pigment flakes (red colour). **a)** Overview of a large flake showing the intact macroscopic structure. **b)** Cross-section revealing the full biofilm thickness and the crystalline organisation of the cells. **c)** High-magnification view of the flake surface, confirming that ordered nanostructure of the rod-shaped bacterial cells is retained within the plane of the film after fixation and mechanical fragmentation.

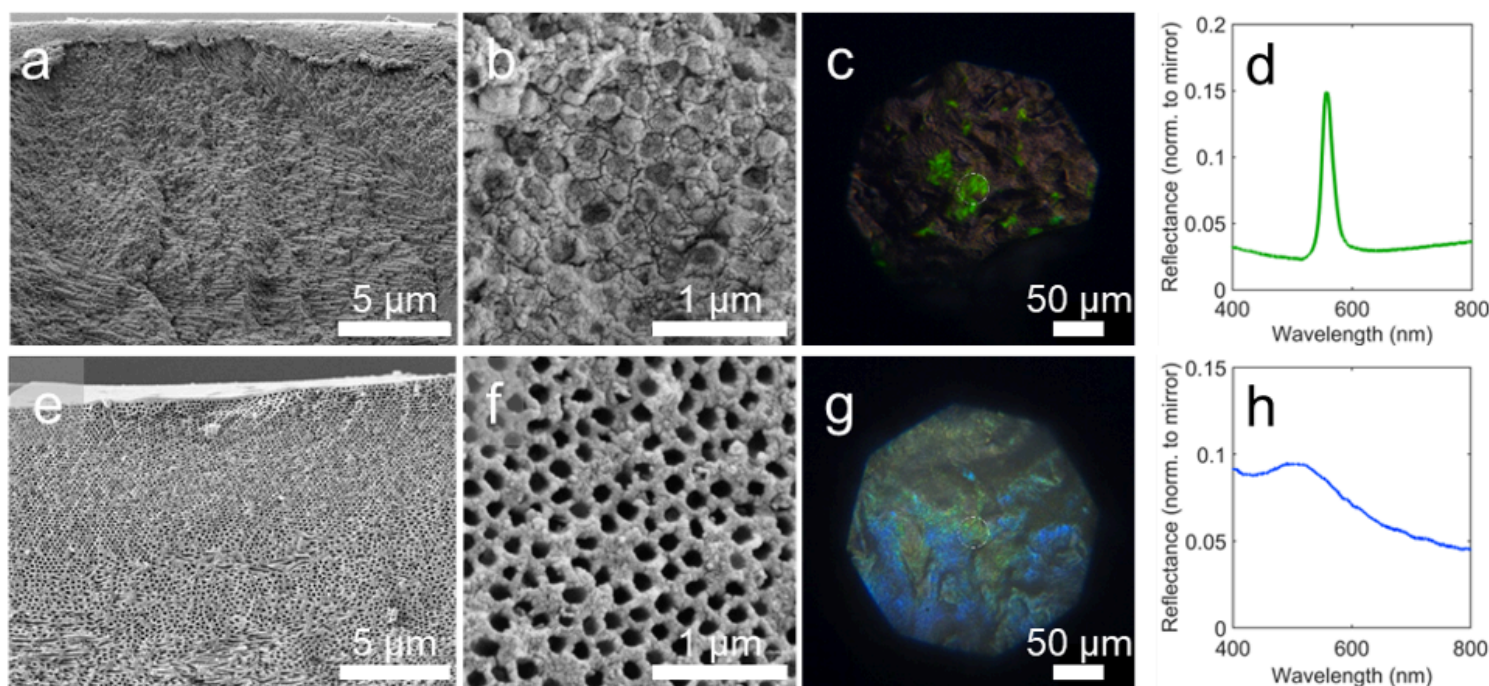


Figure 4

Silica-based fixed biofilms. Optical appearance and morphology of structurally coloured TEOS-infiltrated PlyA2-BC composite biofilms before (a-d) and after the full calcination step (e-h).

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