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Ultra-thin electrodes for wound healing monitoring and drug screening on curved interfaces

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Abstract

Electrical monitoring of wound healing remains limited by insufficient compatibility with curved biological surfaces. Here, we report on ultra-thin, transferable gold electrodes fabricated on poly(2-chloro-p-xylylene) (Parylene C) membranes and integrated with fluorosilicone curved contact lenses, demonstrating their functionality in real-time electrical monitoring of epithelial healing. The platform enables *in vitro* culture of HEK-293 monolayers structurally mimicking curved epithelial models. Healing dynamics are probed by recording the plateau current across pairs of front-facing electrodes under low-amplitude voltage steps, capturing changes in ionic resistance across wound gaps. The system resolves time-dependent restoration of barrier integrity and detects pharmacologically induced modulation with lidocaine, neomycin, and ouabain. Finite element 3D simulations confirm sensitivity to conductivity changes in biologically relevant regimes. This approach offers a geometrically versatile, optically accessible, and drug-responsive interface for dynamic bioelectrical assays on non-planar surfaces, relevant to wound healing and future *ex vivo* corneal regenerative studies.

Introduction

Cell migration and restoration of epithelial integrity following injury are widely studied biological processes relevant to multiple physiological systems, from skin and gastrointestinal tract to airways and cornea.¹ Epithelial healing involves a sequence of cellular events, including migration, proliferation, cell-cell junctions formation, and barrier function recovery.² More broadly, failures in migratory processes underlie several pathological conditions ranging from chronic ulcers to fibrotic diseases and cancer.^{3, 4} Therefore, *in vitro* and *ex vivo* models are being sought for monitoring tissue repair and screening therapeutic agents. Traditional *in vitro* wound healing assays include scratch assays, Boyden chambers, and transwell migration models.^{5, 6} The scratch assay remains widely used due to its simplicity and scalability, involving the mechanical disruption of a cell monolayer, followed by wound closure monitoring over time *via* optical microscopy.^{7, 8} These methods offer visual and quantitative insights into cell migration and proliferation, but rely on optical imaging, are inherently time-discrete, and do not provide continuous real-time resolution of the physical or functional integrity of the monolayer or tissue.⁶

To overcome these limitations, bioelectronic approaches have been developed based on impedance spectroscopy,⁹ transepithelial electrical resistance (TEER),¹⁰ or capacitive sensing^{11, 12}, providing label-free and real-time data on cell coverage, junction tightness, and barrier permeability. Electric cell-substrate impedance sensing (ECIS) and sister technologies have also been used extensively to study cell migration dynamics and response to drugs.^{13, 14} However, these systems are implemented on planar substrates, which do not capture the curvature of native epithelia such as the cornea, the skin, the heart, or the brain.^{15, 16} Significant advances in soft lithography, ultra-thin electronics, and advanced biomaterials have enabled bioelectrical interfaces with higher mechanical compliance and optical transparency.¹⁷⁻¹⁹ Polymeric materials such as poly(2-chloro-p-xylylene) (Parylene C), SU-8, and polydimethylsiloxane (PDMS) have been explored to allow flexibility in conformable devices.^{19, 20} The curvature of biological surfaces is being increasingly considered during wearables design²¹⁻²³ due to its crucial role in modulating cell behavior, including migration orientation, stress distribution, junctional organization, and fluid transport.^{24, 25} Nevertheless, only few wound healing studies incorporate curvature as a variable (**Table S2**).

This work addresses this gap by deploying a transferable and ultra-thin electrode matrix fabricated on flexible Parylene C membranes. We demonstrate feasibility on a commercial fluorosilicone acrylate contact lens, where a HEK-293 cell monolayer is cultured. The key advantages of the proposed work include (i) real-time electrical monitoring of epithelial healing; (ii) electrodes fabrication on ultra-thin Parylene C films, allowing conformal integration onto curved substrates; (iii) pharmacological screening capability, demonstrated by the detection of healing time variations under drug administration; (iv) multiphysics model to predict plateau current variations during wound healing on curved surfaces. Furthermore, the system is fully transparent in the wound region, allowing optical access for simultaneous biological analyses.

Results and Discussion

Ultra-thin, flexible electrode arrays were designed for conformal applications (**Figure 1**). The fabrication process is schematized in **Figure 1a**. Briefly, Corning glass substrates were cleaned in acetone, ethanol, and deionized water, followed by spin coating of a polyvinyl alcohol (PVA) sacrificial layer. A ~2 μm layer of Parylene C was then deposited onto the carrier substrate *via* chemical vapor deposition, forming the flexible membrane. A nominally 100 nm gold film was thermally evaporated onto the Parylene C surface, and the electrodes were patterned by means

of standard photolithography and wet etching. Subsequently, the entire structure was released by dissolving the PVA in water, enabling membrane wet peel-off. The ultra-thin gold-on-Parylene C films were electrically characterized *via* four-point-probes measurements, yielding a sheet resistance of $2.8 \pm 0.5 \, \Omega/\text{square}$. Surface profiles of the Parylene C and ultra-thin gold-on-Parylene C membranes are reported in **Figure S1** and **Table S1**. Validation of the transfer process on a curved geometry is shown in **Figure 1b**. Membranes were re-collected by submerging curved chips in water, and the patterned gold electrodes on Parylene C remained intact under both dry and hydrated conditions. A plexiglass cylinder was then bonded to the final device to facilitate in-well cell culture. **Figure 1c** shows micrographs of the cell monolayer on curved electrodes, demonstrating compatibility of the device with HEK-293 monolayers growth. This process maintained the structural integrity of the cell monolayer over 3 days.

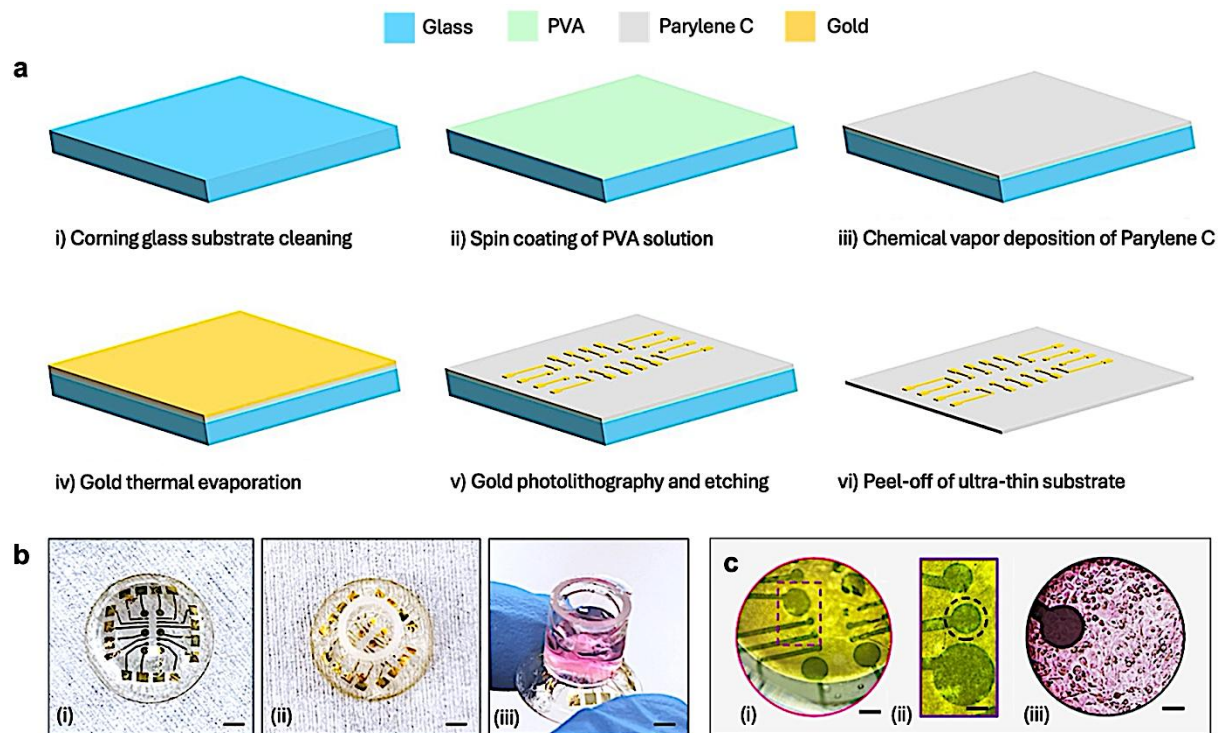


Figure 1. Fabrication and transfer of ultra-thin electrodes. (a) Fabrication of transferrable ultra-thin electrodes: i) glass chip is cleaned in ethanol; ii) spin coating of PVA solution; iii) chemical vapor deposition of Parylene C; iv) thermal evaporation of gold; v) gold photolithography and etching; vi) peel-off of the ultra-thin electrodes on Parylene C. (b) Ultra-thin electrodes on Parylene C membranes are transferred onto curved fluorosilicone acrylate substrates: i) top view of the resulting curved chip; ii) plexiglass well bonding via PDMS dip coating and curing; iii) image of the full platform after cell culture. Scale bars: 2.5 mm. (c) Optical images of the cultured chips: i) snapshot of a curved electrode matrix; ii) snapshot of an array of electrodes; iii) zoom in on the confluent cell monolayer on the chip. Scale bars, left to right: 500 μm .

To evaluate the biocompatibility of the curved geometries, a PrestoBlue cell viability assay was conducted comparing the curved microelectrodes against three substrates commonly employed in biotechnology: a Si_3N_4 membrane, a SiO_2 (glass) chip, and a Ti-Au-coated silicon chip (**Figure S2**). The assay monitors the metabolic reduction of resazurin into resorufin by viable cells, providing a direct colorimetric indicator of cell health²⁶. Quantitative analysis shows that the corrected absorbance values for these groups remain in the range 0.39-0.45. No statistically significant difference was observed between curved electrodes and SiO_2 or Ti-Au substrates (one-way ANOVA with Tukey post-hoc, $p=0.56$ and $p=0.81$; **Figure S2**), demonstrating that the materials and the curved architectures does not disrupt normal metabolic

proliferation. To establish reference pharmacological effects on epithelial migration, a time-lapse live-cell imaging wound healing assay was performed (**Figure 2**). Confluent HEK-293 monolayers were cultured on 96-well plates, mechanically wounded, and assessed over a 34-hour period under drug-free conditions or in the presence of lidocaine, neomycin, or ouabain at varying concentrations. **Figure 2a** shows representative phase-contrast images of wound closure across four selected conditions (drug-free, lidocaine 600 μM , neomycin 3 μM , and ouabain 4 μM). In the absence of treatment, monolayers exhibited continuous and nearly complete wound closure by 34 hours, with wound area visibly reduced by 12 hours. Lidocaine yielded closure comparable to or slightly faster than control at all concentrations ($\sim 70\%$ at 34 h), in accordance with previous studies²⁷, while Neomycin showed a more irregular response, delaying closure at 3.0 μM , but 1.5 μM and 5 μM yielding outcomes comparable with control, consistent with neomycin concentration-specific modulation of phosphoinositide signaling²⁸ and cytoskeletal dynamics²⁹. Ouabain-treated cells showed early signs of cell detachment ($t \sim 6\text{h}$), as previously reported³⁰. The percentages of healed area over time across multiple concentrations for each drug and three wells per conditions are shown in **Figure 2b**.

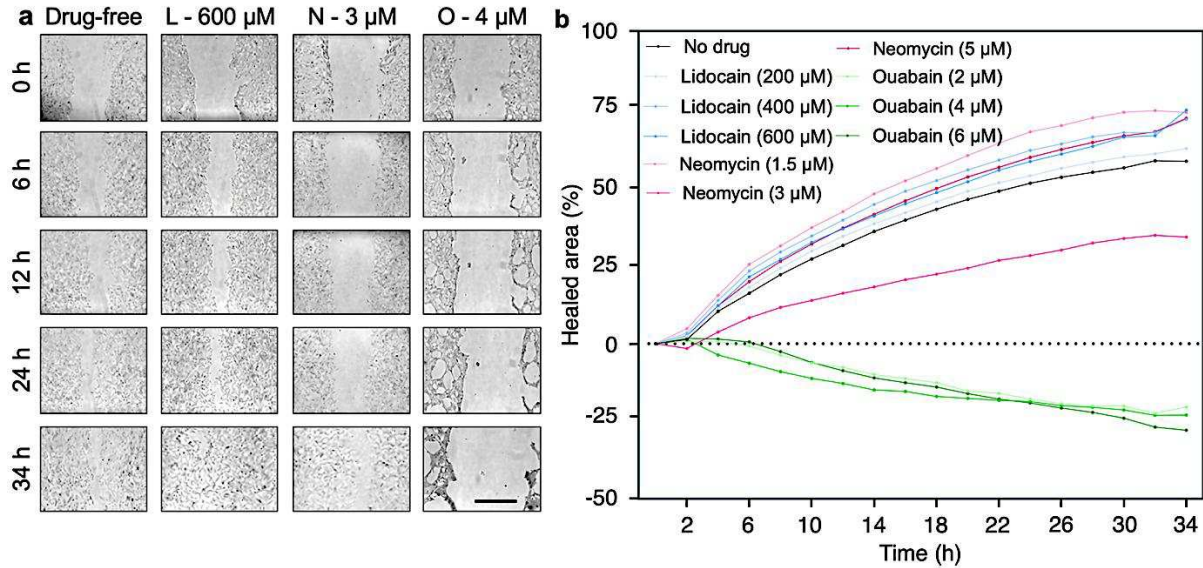


Figure 2. Live-cell imaging of pharmacological effects on epithelial wound healing. (a) Time-lapse phase-contrast images show wound closure over 34 hours under drug-free conditions and in the presence of lidocaine (600 μM), neomycin (3.0 μM), and ouabain (4.0 μM). Scale bar: 500 μm . (b) Wound closure kinetics plotted as percentage of healed area over 34 hours.

The devices were then deployed to monitor the dynamic re-establishment of the epithelial barrier by measuring the plateau current I_{plateau} in response to periodic voltage steps of 10 mV amplitude and 100 ms duration at a frequency of 5 Hz and a duty cycle of 50% (**Figure 3**). **Figure 3d** illustrates the time-resolved equivalent circuits. Immediately after wound induction ($t = 0$), the loss of cellular coverage generated a low resistance setting for lateral ionic current through the exposed culture medium between adjacent electrodes, resulting in a high I_{plateau} . As a first-order approximation, the plateau current can be expressed as:

$$I_{\text{plateau}}(t = 0) \approx \frac{V_0}{R_{\text{wound}}(0) + R_{\text{series}}} \quad (1)$$

where R_{series} accounts for the approximately time-invariant contributions of the electrode-electrolyte interfaces and other series resistances, while R_{wound} represents the effective

resistance of the wound region, at $t=0$. Since the uncovered region is fully exposed to the medium, R_{wound} is minimal, and the current is maximized.

During healing ($0 < t < 24$ h), a decrease in $I_{plateau}$ was attributed to a progressive migration of the cells into the wound gap: this process reduced the area available for direct ionic conduction through the cell-free medium and introduced additional impedance associated with cell-substrate and cell-cell interfaces, while the capacitive components of the system contributed minimally to the current measured within the selected plateau interval. Accordingly:

$$I_{plateau}(0 < t < 24 \text{ h}) \approx \frac{V_0}{R_{wound}(t) + R_{series}} \quad (2)$$

This phase was marked by changes in current over time from ~ 0.9 nA to ~ 0.1 nA, suggesting modulation of barrier properties. By $t \geq 24$ h post-injury, $I_{plateau}$ reached minimum values below 0.05 nA, indicating substantial electrical sealing of the wound region, consistent with reconstitution of the epithelial barrier. Quantitative analysis over 36 hours post-wounding ($n = 3$ independent cultures) confirmed a progressive decrease from 0.89 ± 0.04 nA (mean $\pm$ s.d.) to values below 0.05 nA from 24 h (**Figure 3d**).

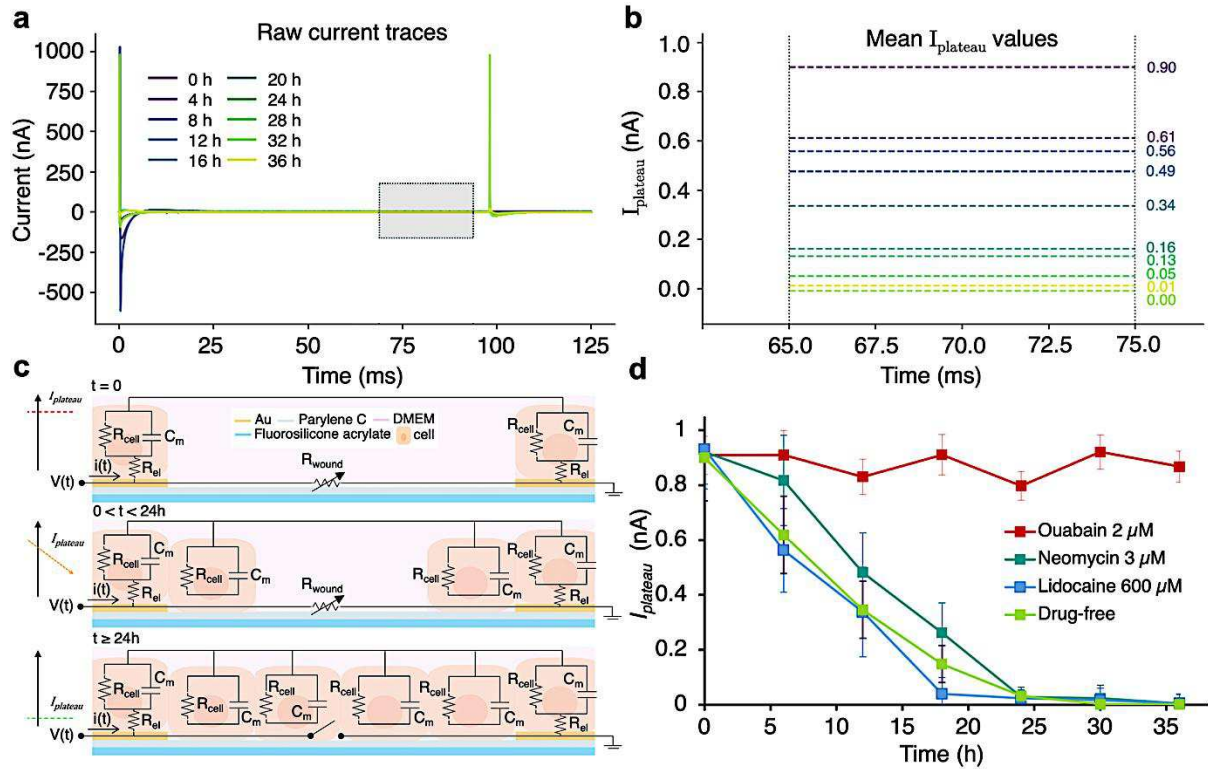


Figure 3. Wound healing monitoring and pharmacological modulation via ultra-thin electrodes on the curved geometry. (a) Representative raw current traces in response to a 10 mV, 100 ms voltage step shown every 4 h over 36 h post-wounding. The shaded box marks the plateau window. (b) $I_{plateau}$ extracted as the mean current in the 65–75 ms window (dotted lines), decreasing from 0.90 nA at 0 h to ≤ 0.05 nA from 24 h onwards. (c) Equivalent circuits in the region of interest at wound induction ($t = 0$), cell migration ($0 < t < 24$ h), and barrier recovery ($t \geq 24$ h). The open switch at $t \geq 24$ h denotes $R_{wound} \gg R_{series}$. (d) $I_{plateau}$ over 36 h for drug-free (light green), ouabain 2 μ M (red), neomycin 3 μ M (green), and lidocaine 600 μ M (blue) conditions (mean $\pm$ s.d., $n = 3$ independent cultures).

Representative raw traces and the corresponding plateau values (65–75 ms window) are shown in **Figure 3a–b**. This current corresponds to the steady-state value after the initial capacitive transient has decayed and reflects the resistive properties of the electrode-cell interface. To

further validate the platform, we treated wounded monolayers with pharmacological inhibitors targeting membrane transport and cytoskeletal pathways^{26, 29}. **Figure 3c** shows the time evolution of I_{plateau} for three representative concentrations previously tested in the high-throughput assay (see *Figure 2*). Cultures treated with 2 μM ouabain exhibited minimal deviation from the wound current levels over 36 hours, maintaining I_{plateau} at nearly constant values (0.88 ± 0.06 nA) due to cell detachment. Treatment with 3 μM neomycin yielded a small delay in current decay, implying that neomycin does not interfere with cellular migration, adhesion, or junction formation required for full electrical sealing at the tested concentrations. The stronger delay seen optically likely reflects the larger 500 μm wound in the planar assay versus the ~ 200 μm gap probed electrically. 600 μM lidocaine produced a faster decay at 18 h (0.03 vs 0.15 nA, drug-free), consistent with the faster closure in *Figure 2b*.

A 3D finite element model provided insights on the electrical behavior of the curved system (**Figure 4**). **Figure 4a** shows a side view of the modeled material stack, also reported in Methods. The conductivity of the wound region was modeled as linearly decreasing from a starting value of $6 \times 10^{-3} \text{ S m}^{-1}$ to $3.3 \times 10^{-4} \text{ S m}^{-1}$ as cells progressively covered the wound area. These values were selected as representative of the experimental conditions, considering the 0.9 nA and 0.05 nA measured plateau currents ($R_{\text{total}} = 11.1 \text{ M}\Omega$ and $R_{\text{total}} = 200 \text{ M}\Omega$ at 0% and 100% wound progression). The computational model yielded plateau currents ranging from 2.3 to 49 nA (**Figure 4b**). Despite this discrepancy in absolute magnitude attributed to non-ideal effects in the experimental setup, the simulated trend shows a progressive linear decrease and a 94% I_{plateau} reduction until full barrier restoration, mirroring the monotonic decay of recorded experimentally and confirming that the electrode pair operates in a regime where the readout is dominated by the ionic resistance of the wound region rather than by interfacial or geometric contributions.

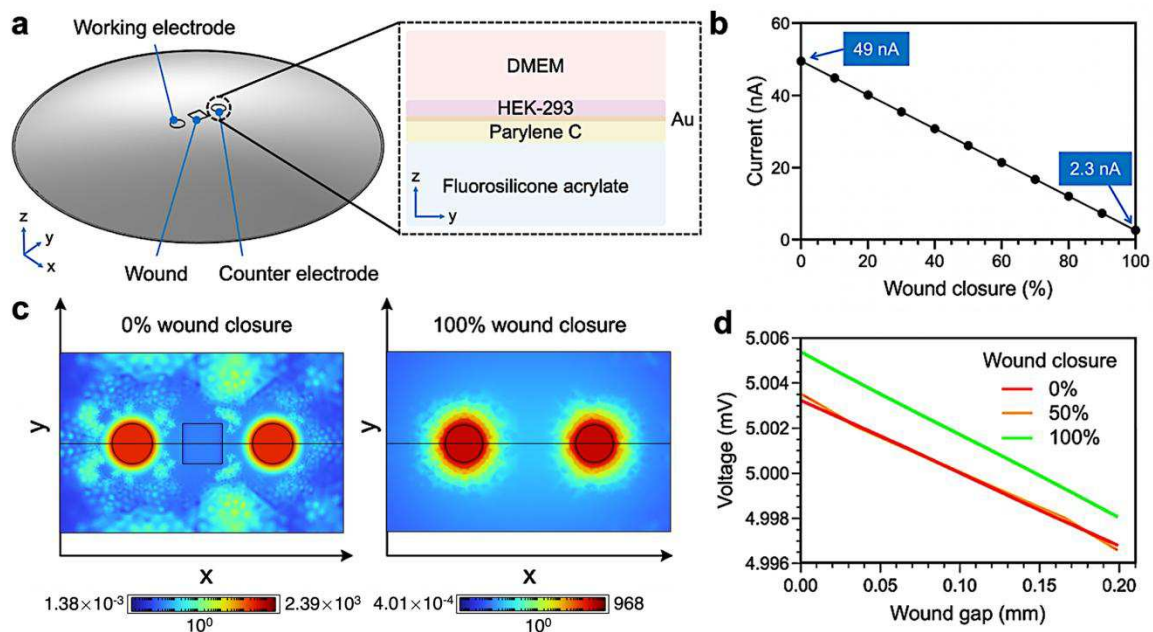


Figure 4. Finite element modeling of the curved bioelectronic platform. (a) 3D geometry of the curved fluorosilicone acrylate substrate carrying a working and a counter gold electrode (100 μm radius, 500 μm apart) with the wound region located in between. The inset shows the side view of the modeled material stack. (b) Simulated current between the working and counter electrodes as a function of wound closure (0-100%), yielding a decrease from 49 nA to 2.3 nA. (c) Surface plots of the electric field norm (V m^{-1}) in the electrode plane at 0% and 100% wound closure under a 10 mV bias, showing field concentration at the electrode edges and the low-

field wound gap prior to closure. **(d)** Voltage profiles along the line connecting the two electrodes across the 200 μm wound gap at 0%, 50%, and 100% wound closure.

Surface plots of the electric field norm (**Figure 4c**) show that the field is concentrated at the electrode perimeters ($5\text{-}10\text{ V m}^{-1}$), consistent with edge current crowding on circular electrodes, while the exposed wound at 0% closure appears as a low-field region between the electrodes. Upon full closure, the low-conductivity cell layer redistributes the field around the electrode rims. Voltage profiles extracted along the line connecting the electrodes across the wound gap (**Figure 4d**) remained quasi-linear around the 5 mV midpoint, with a drop of $\sim 7\text{ }\mu\text{V}$ over 200 μm and a progressive upward shift with closure, indicating that the electrode-to-wound path becomes increasingly resistive as cells repopulate the gap.

Conclusion

The functionality of freestanding electrode membranes transferred onto unconventional geometries was demonstrated by monitoring barrier restoration via current measurements in HEK-293 cell monolayers cultured on a curved substrate. Healing dynamics were monitored by probing the amplitude and the temporal evolution of I_{plateau} values across front-facing electrodes. Electrode size and pitch defined the operating regime in terms of electrical signal and biological specificity. The 100 μm size was found to be large enough to yield measurable current, and small enough to preserve sensitivity to cell coverage. The measurement protocol based on low-amplitude and low-frequency voltage steps ensures electrochemical stability of the gold interface and avoids Faradaic interference, allowing non-destructive monitoring of epithelial barrier recovery. The system offers a label-free approach to resolve biophysical and pharmacologically induced modulations of the healing process *in vitro*, with the potential to be translated to *ex vivo* corneal tissues in follow-up studies.

Materials and Methods

Fabrication, characterization, and transfer of flexible electrodes. The fabrication began with standard 3-inch Corning borosilicate glass wafers, cleaned by immersion in acetone, isopropanol, and deionized water, each with 5 minutes of sonication, followed by drying with a nitrogen gun. A 5 wt% aqueous solution of polyvinyl alcohol was spin-coated onto the clean glass substrate at 3000 rpm for 60 seconds to form a uniform sacrificial layer. The polyvinyl alcohol-coated substrates were baked at 80 $^{\circ}\text{C}$ for 10 minutes to drive off water and ensure adhesion. Further, a $\sim 2\text{ }\mu\text{m}$ -thick layer of Parylene C was deposited using via chemical vapor deposition, followed by deposition a 100 nm-thick layer of gold by thermal evaporation under high vacuum ($<1 \times 10^{-6}$ Torr) at a rate of $1\text{ }\text{\AA s}^{-1}$. Gold was patterned by spin-coating a positive photoresist (S1813, Microposit) at 4000 rpm for 60 seconds, followed by a soft bake at 95 $^{\circ}\text{C}$ for 90 seconds. UV exposure was performed through a chrome mask using a mask aligner (EVG620), followed by development in MF-319 for 60 seconds. The exposed gold was etched in a potassium iodide/iodine-based etchant until clear pattern definition was achieved. The remaining photoresist was stripped in acetone. The gold-on-Parylene C films were electrically characterized in air by means of 4-point-probes measurements (Ossila Four-Point Probe). Parylene C and Gold thickness characterization was performed using a mechanical profilometer (Dektak XT by Bruker). To release the electrode membrane, the glass support was immersed in deionized water for 2 minutes until the polyvinyl alcohol layer fully dissolved. Floating devices were wet transferred onto curved substrates. Curved substrates were previously cleaned *via* Oxygen plasma treatment (100% O_2 , 100W, 180 s). Gold adhesion to Parylene C was maintained throughout transfer, and electrical continuity of the electrodes was confirmed via multimeter probing after processing.

Assessment of electrode stability. The electrical characterization of the Au/Parylene C electrodes was assessed by repeated impedance measurements during 48 h of immersion in PBS. Impedance magnitude and phase angle were measured using an RS PRO LCR-6300 precision LCR meter in Z- θ mode over a frequency range of 10 Hz–300 kHz. Measurements were performed using a 10 mV AC excitation, with no applied DC bias. The complete temporal evolution was recorded for a representative electrode, while measurements from five electrodes were used to calculate mean values and standard deviations at selected time points. These results are reported in **Supplementary Figures S3-S4**. Optical microscopy images were acquired using an OKM 173 trinocular metallurgical microscope, equipped with a 1 $\times$ C-mount adapter (OBB-A1514) and an ODC 832 5.1-megapixel USB 3.0 microscopy camera (**Supplementary Figure S5**). The electrode surface was imaged before and after 48 h of immersion in PBS to assess optically detectable alterations, including cracking, blister formation, peeling, or delamination of the Au film.

Presto blue viability assays. Cells were cultured into dedicated well plates for PrestoBlue assay. PrestoBlue Cell Viability Reagent was added to each well (1:10). All samples were incubated at 37°C with 5% CO₂ for 1-3 hours to allow for metabolic activity-induced colorimetry. The solutions were then transferred to a multiwell plate for spectrophotometry absorbance measurements at 570 nm and 600 nm, where the latter acts as the reference wavelength. Data was analyzed by (i) blank correction: the average absorbance of the blank control was subtracted at both 570 nm and 600 nm from the corresponding absorbance readings of each experimental group, and (ii) absorbance correction: the corrected absorbance at 570 nm was calculated by subtracting the corrected absorbance at 600 nm from the absorbance at 570 nm. Higher normalized absorbance values indicate higher metabolic activity and suggest better biocompatibility of the substrate.

Drug preparation. Lidocaine was dissolved in sterile deionized water to yield 100 mM stock solutions. Working concentrations (200, 400, and 600 μ M) were obtained by dilution in complete cell culture medium (DMEM high glucose + 10% FBS + 1% penicillin/streptomycin) immediately before use. Neomycin sulfate was prepared in 100 mM aqueous stock solutions, and working concentrations (1.5, 3.0, and 5.0 μ M) were diluted into cell culture medium prior to each experiment. Ouabain octahydrate was dissolved in sterile deionized water at 10 mM and working solutions (2.0, 4.0, and 6.0 μ M) were prepared by dilution into cell culture medium before use. For all experiments, drugs were added after mechanical wounding of the HEK-293 monolayers.

Cell migration assay and live imaging. HEK-293 cells were seeded cultured in 96-well plates at a density of 1×10^5 cells/well to achieve a confluent monolayer within 24 hours. At confluency, 500 μ m wounds were scratched in individual wells using a 10 μ L tip. Further, wells were gently washed with sterile PBS to remove detached cells and then replenished with fresh culture medium, with or without pharmacological agents. The plate was continuously imaged using Omni (Axion Biosystems, Inc., USA) from inside the incubator, at 37°C and 5% CO₂. Phase-contrast images were acquired every 2 hours for a total of 34 hours. Wound healing data was collected from the dedicated software and plotted to represent the percentage of the initial wound area occupied by migrating cells. Data were calculated as the mean of 3 wells per tested condition.

Cell culture on curved chips. Prior to seeding, curved devices with bonded wells as were cleaned under low O₂ plasma treatment (30 W, 120 s, 100% O₂), sterilized for 30 min under UV light, and coated with 1 mg mL⁻¹ poly-L-lysine for 45 min to facilitate cell adhesion. Prior to culture, devices were washed in three steps using deionized water. Curved devices were then

air-dried for 15 min. HEK-293 cells were seeded at a density of 120k cells/well and maintained in DMEM high glucose, supplemented with 10% FBS and 1% penicillin/streptomycin. Within 24 hours, cells adhered uniformly and reached confluency. Throughout the culture period, no detachment was observed. Wounds were generated by a single manual pass of a sterile pipette tip across the inter-electrode gap, perpendicular to the electrode axis, yielding ~200 μm -wide wounds confined between the two electrodes (geometry used in the FEM, Figure 4). Detached cells were removed by two PBS washes.

Electrical measurements of wound healing and drug screening. Electrical measurements were performed by connecting pad pairs to the two-probes low-noise current amplifier Axopatch 200B (Axon Instruments, Inc., USA), configured for time-resolved current measurements under voltage-controlled stimulation. A square wave voltage input (10 mV, 100 ms) was applied between each electrode pair, at a frequency of 5 Hz. The plateau current (I_{plateau}), defined as the steady-state current reached after the initial capacitive transient in each cycle, was recorded and averaged over 5 cycles per time point to reduce noise. Data were acquired at a sampling rate of 10 kHz, sufficient to resolve transient and steady-state phases, using pCLAMP 11 software suite. Baseline measurements were recorded prior to wounding to confirm monolayer confluency and electrode coverage. After wounding, the medium was replaced with fresh culture medium with or without pharmacological agents. Measurements were performed every 3 to 6 hours over a total period of 36 hours. All measurements were performed on an optical table and a custom Faraday cage using shielded cables and grounded instrumentation to minimize electrical artifacts. All recordings were acquired from the central pair of 100 μm electrodes (500 μm pitch), additional geometries on the mask served as transfer-yield test for non-planar microelectrode arrays structures and were not used.

Finite element modeling. A 3D stationary finite element simulation was performed using COMSOL Multiphysics®6.2. The geometry comprises a curved fluorosilicone acrylate substrate with a Parylene C layer on top. Working and counter electrodes are represented as circular gold layers with radius of 100 μm and thickness of 100 nm, symmetrically placed 500 μm apart with respect to the center. A 10 μm layer of HEK-293 cells was modeled on top of the electrodes, within the cell culture medium. The wounded volume is represented as a square gap with 200 μm side between the two electrodes. The stationary electric field distribution was obtained by solving the electric current physics interface, under a continuous bias of 10 mV applied between the working and counter electrodes. All external boundaries surrounding the medium and the substrate are treated as insulating. The computational domains are discretized using a user-defined unstructured mesh with free tetrahedral and triangular elements. For each healing stage, electric field distributions were visualized using surface and vector plots, and voltage profiles were obtained by extracting values along a line connecting the two electrodes across the wound region. The current between the working and the counter electrode is also evaluated for different healing stages.

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Data availability statement

All datasets are available from the corresponding authors upon request.

Author contribution statement

FAV fabricated and characterized the devices. RM performed electrical measurements. CS and CDM supported cell culture. CFJ run the biological assay. MV built the 3D computational model supervised by CC. DI built a preliminary 2D computational model supervised by AMP. CB, MTR, and TP provided resources. RM conceived and led the project, and acquired funding.

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