

A programmable bioresorbable electrochemical microneedle sensor array for perioperative monitoring of organ health

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Comprehensive and continuous assessment of organ physiology and biochemistry, beyond the capabilities of conventional monitoring tools, can enable timely interventions for perioperative complications such as organ ischaemia and transplant rejection. Here we present an integrated bioresorbable system that enables multiplexed, real-time and spatially mapped electrochemical monitoring of deep organs throughout the surgical course. Using a 3D printing-based, photolithography-free fabrication process, the system features a flexible, 3D programmed, individually addressable microneedle sensor array with backward-facing barbs for conformal and stable organ interfacing and 3D parenchymal probing. Electrochemical functionalization of microneedle tips enable concurrent monitoring and spatial mapping of key biochemical markers, such as electrolytes, metabolites and oxygenation, in deep organs for at least 7 days. An electrically programmable self-destruction mechanism offers controllability over the degradation process, eliminating the need for device retrieval. Demonstrations in clinically relevant complications such as kidney ischaemia and gut disorders in animal models highlight the broad applications of this device in intra- and postoperative monitoring, advancing perioperative care and critical care medicine.

Comprehensive and continuous monitoring of organ functions throughout the surgical course, from intraoperative evaluation to postoperative recovery, is essential for optimal patient outcomes^{1,2}. Intraoperative complications arising from surgical procedures, anaesthesia, medications and individual patient susceptibilities could lead to severe or even irreversible organ dysfunction that cannot be timely informed by traditional vital sign-based intraoperative monitoring (for example, electroencephalography (EEG), electrocardiography (ECG), blood pressure (BP) and oxygen saturation (SpO₂))^{3,4}. Examples include acute kidney and liver injuries due to ischaemia and/or

nephron-/hepatotoxic agents, and bladder dysfunction due to accidental nerve damages^{5,6}. On the other hand, capabilities in monitoring organ conditions in the postoperative phase, including potential surgical complications (for example, anastomotic leak after gastric surgeries)⁷, restoration of organ functions (for example, bladder control after partial cystectomy)⁸, transplant rejection (for example, kidney and liver)^{9–11} and stress responses to surgery (for example, metabolic disorders, gut dysmotility)¹², are crucial to the recovery of patients. The first few postoperative weeks represent the most critical window, during which patients are at the highest risk for major surgical

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complications¹³. These complications often develop insidiously, and conventional methods such as blood tests often lack specificity and show delayed responses to localized organ conditions¹⁴. Radiological tests, while valuable, could only be performed intermittently in specialized facilities^{15,16}. Consequently, these conditions often remain undetected until after the development of severe outward symptoms, leading to substantial increases in morbidity and mortality¹⁷. These challenges underscore a critical, unmet clinical need for comprehensive and continuous assessment of organ functions to facilitate timely interventions during both intra- and postoperative phases¹⁸.

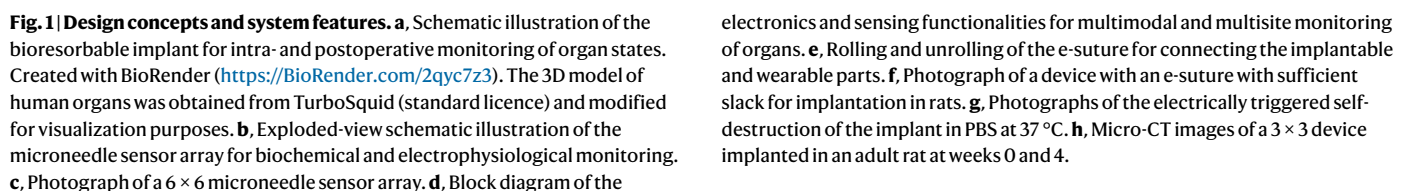
Biochemical markers serve as highly sensitive and specific indicators of organ dysfunction and disease¹⁹. Advances in bioelectronics and biosensors have driven considerable progress in developing devices for biochemical monitoring of relatively shallow or accessible body regions²⁰. Examples include skin-mounted microneedle devices for tracking subcutaneous metabolites and electrolytes^{21–24}, catheter- and fibre-based devices for monitoring pharmaceuticals and metabolites in superficial veins^{25–28}, and miniaturized probes for detecting ions and neurotransmitters in the brain^{29,30}. In contrast, developing devices for biochemical monitoring in deep organs, such as the kidney and liver, presents substantial challenges because of (1) the need for miniature, soft and highly mechanically compliant form factors that can access, interface with, probe and affix to deep organs without causing irritation or undue burden on the body; (2) the requirement for chronic functionality, as frequent replacement is impractical; and (3) the necessity of careful consideration of their eventual clearance from the body. Existing deep-organ monitoring devices^{31–33}, however, exhibit substantial limitations in these regards: (1) They typically have rigid and/or bulky form factors due to the presence of batteries and circuit boards, which can irritate surrounding tissue and dislodge over time. (2) They primarily monitor biophysical parameters (temperature, pressure, impedance, oxygenation and so on) and lack biochemical monitoring capabilities^{31–34}, not to mention their multiplexed and chronic monitoring. (3) They typically require secondary surgery for device retrieval, posing additional risks and clinical burdens. Thus, devices that can access, interface with, probe and affix to deep organs with minimal irritation, chronically monitor a multiplexed panel of biochemical markers and eliminate the need for surgical retrieval represent an unrealized ideal in deep organ monitoring.

Towards addressing the challenges of deep organ interfacing and biochemical detection, microneedles hold great potential by being able to minimally invasively access and probe organ parenchyma. Extensive methods, including microfabrication³⁵, micromachining³⁶, three-dimensional (3D) printing³⁷, micromoulding³⁸ and hybrid approaches³⁹, have been developed for fabricating microneedle arrays, with recent advances that combine rigid microneedles with flexible substrates to enable conformal interfacing with the human body⁴⁰. Primarily developed for skin-mounted uses, microneedle sensors have enabled high-quality recording of electrophysiology (for example, electromyography^{41–43}) and accurate monitoring of biochemical markers in the subcutaneous space when electrochemically functionalized^{21,23}. Despite such progress, existing fabrication methods for flexible microneedle arrays still have substantial limitations for monitoring deep organs: (1) Microfabrication enables the fabrication of highly miniaturized and sophisticated patterns but is costly, labour-intensive and reliant on sophisticated equipment⁴¹. In addition, as a planar fabrication technology, it is not suitable for deep (~mm) and sophisticated vertical structures, and does not offer programmability of microneedle lengths across the array, limiting the ability to target organ regions of varying depths. (2) 3D printing and micromachining, often combined with micromoulding, allow for relatively simple and low-cost fabrication of 3D programmed microneedle patterns. However, they do not support individual addressability of microneedles that would otherwise require photolithography-defined metal patterning or sophisticated multimaterial printing strategies^{23,40},

disallowing spatially resolved mapping of organ states across different functional regions. (3) Conventional microneedles show poor tissue adhesion, making stable affixation to organs challenging²⁸. Recent development of microneedles with swellable tips⁴⁴, tilted angles⁴⁵ and backward-facing barbs^{46–48} provided enhancement of tissue retention by up to 18 times⁴⁸, but they rely on complex fabrication techniques, including 4D printing⁴⁸, magnetorheological drawing lithography⁴⁶, two-photon photopolymerization⁴⁷, microfabrication⁴⁵ and specialized polymer coatings⁴⁴. Overcoming these limitations is essential for developing microneedle-based deep organ monitoring technologies.

Towards addressing the challenges related to device clearance from the body, bioresorbable electronics has emerged as a promising solution by enabling device disintegration and dissolution in the body after a period of operation⁴⁹. Leveraging various materials and manufacturing strategies (for example, photolithography, 3D printing)^{50,51}, a diverse range of bioresorbable devices have been developed for delivering stimulations and drugs^{52,53}, monitoring physical parameters such as strain, pressure, temperature, flow and biopotential^{51,54,55}, and a limited set of electroactive chemicals such as dopamine and nitric oxide^{56,57}. However, existing bioresorbable electronics remain limited in their capabilities for biochemical monitoring in deep organs: (1) Electrochemical detection relies on a standard, non-bioresorbable three-electrode system, comprising a working electrode (WE, typically Au), a reference electrode (RE, typically Ag/AgCl) and a counter electrode (CE, typically Pt). While recent work based on galvanic coupling of bioresorbable metals enabled a bioresorbable three-electrode system for glucose monitoring, challenges such as potential drift and the need for a complex signal compensation module limit its practical applicability⁵⁸. (2) Current bioresorbable devices lack controllability over their degradation process, as they follow natural bioresorption processes with limited tunability. (3) Electrochemical sensing relies on a potentiostat chip, which is not bioresorbable. While chip-less readout schemes, such as ultrasound and inductor–capacitor (LC) circuits^{59,60}, enabled fully bioresorbable systems for select parameters, they are not generalizable for electrochemical measurements that require sophisticated interrogation waveforms. An alternative that has been explored is to externalize the electronics and insert the probe percutaneously, which, however, is susceptible to probe displacement and wound irritation due to external forces¹¹. Addressing these challenges is critical for advancing bioresorbable electronics for deep organ biochemical monitoring.

Here we report an integrated system that collectively addresses these challenges and offers the capability of chronically monitoring and spatially mapping multiple biochemical markers in deep organs for at least 7 postoperative days and performing electrically programmed self-destruction upon completion of monitoring. Key features and advances include: (1) conformal interfacing, 3D probing and stable affixation to deep organs through a flexible, 3D programmed, individually addressable, barbed and bioresorbable microneedle array enabled by a 3D printing-based, photolithography-free fabrication process; (2) a rolling-based bioresorbable electronic suture (e-suture) with parallel electrical interconnects for connecting the implant and skin-mounted electronics, eliminating the need for electronics implantation while supporting normal wound healing^{61,62}; (3) multiparametric monitoring of electrolytes (for example, Na⁺, K⁺, pH), metabolites (for example, glucose (GLU), uric acid (UA), lactic acid (LA)), tissue oxygenation and electrophysiology, and multisite mapping of up to 32 sites, with antibiofouling strategies that offer at least 7 days of *in vivo* monitoring; and (4) electrically triggered dissolution of electrochemical sensing electrodes that offers degradability of the electrodes and programmable self-destruction of the device after use. Demonstrations in rodent models for monitoring acute and chronic ischaemia in the kidney and metabolic disorders in the gut highlight extensive capabilities of the device for comprehensive and continuous assessment of organ functions in clinically relevant complications.



and wearable parts. The system monitors organ states and streams data in real time to remote monitors in intra- and postoperative settings (Fig. 1a). Upon completion of monitoring, the implant is electrically programmed to initiate self-destruction and gradually bioresorbs in the body, eliminating the need for secondary surgery to retrieve the implant. The bioresorbable implant (Fig. 1b,c) is a flexible microneedle

array consisting of poly(lactic-co-glycolic acid) (PLGA) microneedles and substrate, molybdenum (Mo) electrical interconnects and poly-anhydride (PA) encapsulations, all of which are bioresorbable through hydrolysis (Extended Data Fig. 1a)⁵³. The microneedles are externally coated with electrode materials (Au, Pt, Ag/AgCl) for electrochemical measurement, which can be electrically dissolved to expose the underlying bioresorbable materials and initiate self-destruction. The microneedle sensors are individually functionalized and addressed to measure various biochemical markers, such as GLU, LA, UA, pH, O₂, Na⁺ and K⁺, as well as electrophysiology, such as electromyography (EMG). The microneedle array seamlessly conforms to the organ surface, probes the organ parenchyma in a spatiotemporally resolved manner and anchors firmly to tissue through backward-facing barbs.

As shown in Fig. 1d and Supplementary Fig. 1, the wireless electronics module (footprint: 21 mm × 25 mm) consists of a BLE system-on-a-chip (SoC, nRF52832), an electrochemical front end (AD5941), a 32-channel switch matrix (ADG732), a voltage buffer array (ADA4505) and a small lithium-polymer battery (65 mAh). The 32-channel switch matrix allows for multimodal monitoring of up to 32 amperometric biomarkers (for example, GLU, LA, UA, O₂), multisite mapping of a single biomarker across up to 32 sites, or a combination of these 2 modes. The voltage buffer array allows monitoring of multiple potentiometric biomarkers, such as K⁺, Na⁺, pH and biopotential. The device features low power consumption of ~0.5 mA in idle mode and ~7 mA in active measurement mode (Supplementary Fig. 2).

The electrical interconnects of the microneedle array can be rolled into an e-suture with parallel interconnects (Fig. 1e and Extended Data Fig. 1b)^{61,62}. While surface-coated functional sutures have been developed for sensing strain⁶³, pH⁶⁴, biopotential⁶⁵ and chemicals from gastric leak⁶⁶, they are non-bioresorbable and contain only one or few electrical interconnects, thereby not meeting the needs of this application. The diameter of the rolled e-suture is dependent on the thickness of the substrate and the total width of the parallel interconnects (Extended Data Fig. 1c,d and Supplementary Note 1). With a PLGA substrate of 40 µm thickness, the e-suture for a 3 × 3 microneedle array has a diameter of ~0.5 mm, which corresponds to a standard United States Pharmacopeia (USP) #0 suture typically used for abdominal wall closure and is smaller in diameter than a standard USP #4 suture (Fig. 1e (middle left) and Extended Data Fig. 1e,f). It also uses a standard suture material (PLGA), facilitating seamless use as standard suture (Fig. 1e, middle right). During rolling, the middle segment undergoes thermal treatment at 80 °C, above the glass transition point of PLGA (<60 °C)⁶⁷, to permanently fuse it into a stable structure. The distal segment of the e-suture is not thermally treated, allowing for unrolling and connection to the electronics module (Fig. 1e, right). E-sutures with extended lengths (~10 cm, Fig. 1f) provide slack between the microneedle array and the abdominal wall, preventing displacement caused by abdominal wall movement in small animals. Photographs of completed 3 × 3 and 6 × 6 devices on the palm further illustrate the form factor of the devices suitable for small and large animals (Fig. 1f and Extended Data Fig. 1g,h), respectively.

The encapsulation layer consists of two bioresorbable forms of PA: poly(butaneedithiol 1,3,5-trialyl-1,3,5-triazine-2,4,6(1H,3H,5H)-trione pentanoic anhydride (PBTPA) and poly(ethylene glycol) diacrylate pentenoic anhydride (PEGPA). The hydrolysis rates of PBTPA and PEGPA are ~1.3 µm day⁻¹ and ~27 µm day⁻¹ in phosphate-buffered saline (PBS) at 37 °C, respectively. Simple mixing of the two PAs in solution yields polymer blends with tunable hydrolysis rates⁵³. By using a 50:50 PA mixture, the bioresorbable implant remains visually intact for 4 weeks in PBS at 37 °C which simulates the physiological environment (Extended Data Fig. 1i). The individual electrical interconnects of the e-suture maintain stable electrical resistance during this period (Extended Data Fig. 1j). In comparison, upon triggering self-destruction after week 2, the implant gradually undergoes bioresorption and disappears by week 9, highlighting the programmability of the bioresorption

process (Fig. 1g). Microscale computed tomography (micro-CT) imaging (Fig. 1h, left) shows the device implanted in a rat, with the electronics mounted on the skin. When triggered after week 1, the implanted device degrades rapidly in vivo, becoming undetectable by week 4. These findings collectively demonstrate the device's form factor, functionality, bioresorbability and self-destruction capability, making it suitable for comprehensive and continuous monitoring of organ health throughout the surgical course.

3D printing-based, photolithography-free fabrication of the microneedle array

Figure 2 and Supplementary Fig. 3 illustrate the fabrication process. This process leverages standard 3D printing, with materials and parameters detailed in Supplementary Fig. 4. It offers high programmability of microneedle diameters (≥100 µm), sharpness (radius of curvature: ≥10 µm) and lengths (from submillimetres to millimetres) across the array (Supplementary Fig. 5). However, as a layer-by-layer manufacturing technology, 3D printing does not support the direct manufacturing of microneedles with backward-facing barbs because printing a layer without the support of a preceding layer is impossible. To address this limitation, this work proposes a deformation-coupled 3D printing technique. The process starts with standard 3D printing of microneedles with horizontal barbs and serpentine interconnects (Fig. 2a,b). By default, the microneedles are 300 µm in diameter and 1,000 µm in length, and the barbs are 80 µm in diameter and 400 µm in length. Next, a polydimethylsiloxane (PDMS) mould with cone-shaped patterns compresses the horizontal barbs into pre-designed backward-facing barbs (described in detail in the subsequent section), followed by curing of the 3D-printed resin under 405 nm ultraviolet (UV) light and removal of the PDMS mould (Fig. 2c,d). A double moulding process using Ecoflex then transfers the barbed microneedle patterns from 3D-printed resin to bioresorbable PLGA (Fig. 2e,f). Subsequently, sputtering of Mo (500 nm) on the entire sample (Fig. 2g,h) followed by Cr/Au (10/100 nm) on the microneedles using a paper mask that exposes the microneedles completes the metallization process (Fig. 2i,j). Next, a transfer printing process transfers and bonds the metalized PLGA structure to a thin film of PLGA (20-µm thick) at 80 °C (Fig. 2k,l), followed by covering another PLGA film (20-µm thick) on the top. The entire substrate is immersed in a mould with PA to form a PA encapsulation layer of 100 µm thickness after UV curing at 365 nm. Afterwards, a PDMS slab screens the sensing microneedle tips and self-stops upon reaching the barbs, followed by spin coating a thin layer of PA (~10 µm) to encapsulate the microneedle body (Fig. 2m,n). Next, electrodeposition of gold nanoparticles (AuNPs) using HAuCl₄ solution (see Methods) on the sensing microneedle tips enhances the electrode surface area and facilitates faster electron transfer kinetics (Fig. 2o,p)⁶⁸. The AuNPs are densely packed and/or coalesced, with a diameter of ~20 nm (Fig. 2p, inset). This simple method provides strong adhesion to the electrode compared to colloidal AuNPs⁶⁹, as evidenced by the stable morphology and sensing performance in chronic in vivo animal studies in subsequent sections. Lastly, according to specific sensor functions, functionalization of individual microneedles with sensing and protective coatings (Supplementary Fig. 6) completes the fabrication process.

In vitro and in vivo characterizations of the system

Enhancing the tissue adhesion of microneedles critically relies on the monolithic fabrication of backward-facing barbs, as detailed in Extended Data Fig. 2a. Following deformation of horizontal barbs into backward-facing barbs using a PDMS mould, a moulding process negatively transfers the patterns to Ecoflex. After temporarily bonding the Ecoflex mould to a glass slide to form an enclosed structure, PLGA melts and infiltrates into the mould in a vacuum oven (160 °C), replicates the patterns from 3D-printed resin to PLGA. The low modulus of Ecoflex is crucial for the high-fidelity transfer of patterns with

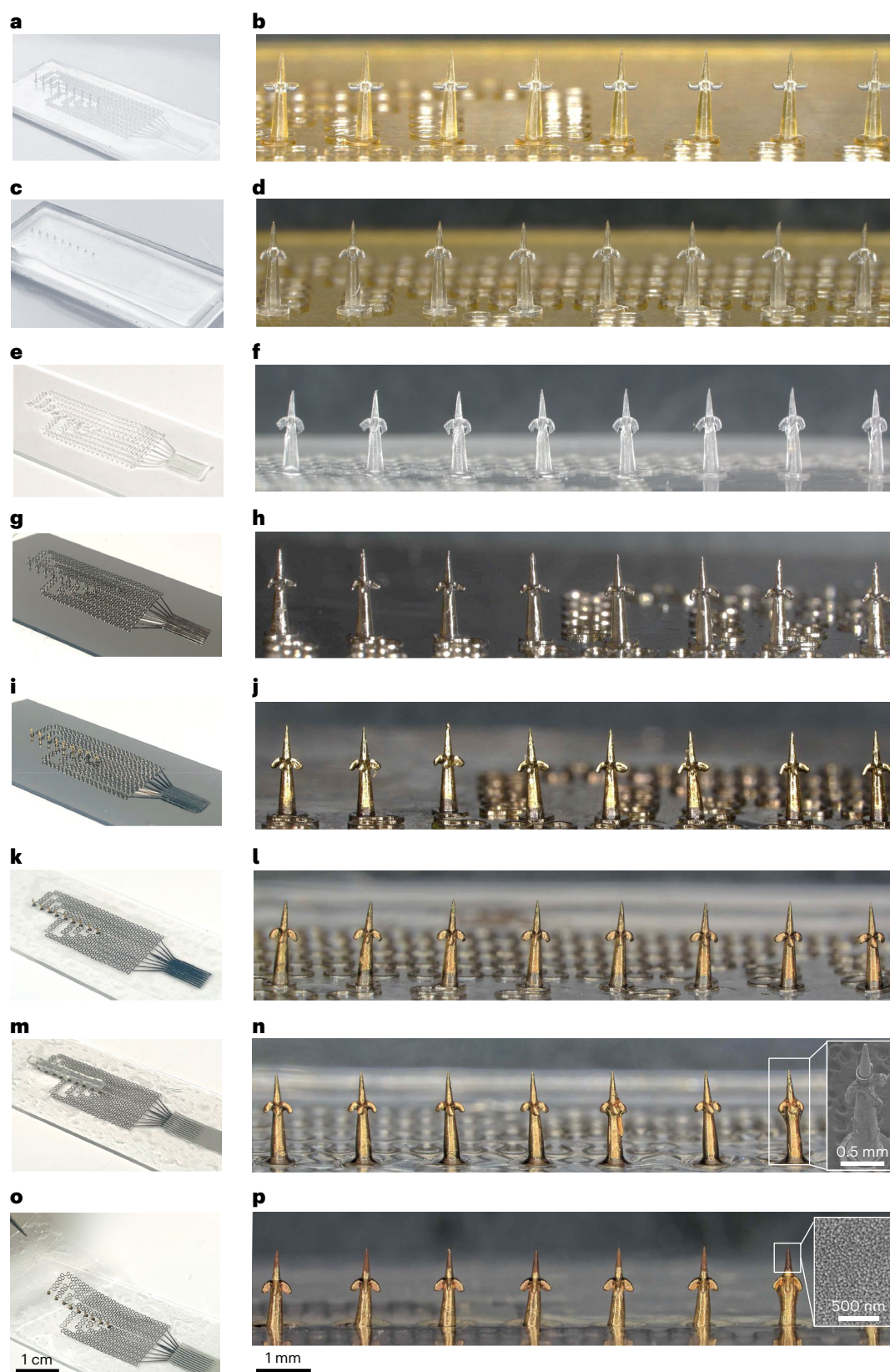


Fig. 2 | Photographs and micrographs of the 3D printing-based, photolithography-free fabrication process. a,b, Photograph (a) and magnified view (b) of the 3D-printed mould. **c,d,** Photograph (c) and magnified view (d) of the barb deformation process using a PDMS mould. **e,f,** Photograph (e) and magnified view (f) of the PLGA microneedle array. **g,h,** Photograph (g) and

magnified view (h) after Mo sputtering. **i,j,** Photograph (i) and magnified view (j) after Au sputtering. **k,l,** Photograph (k) and magnified view (l) after bonding to PLGA substrates. **m,n,** Photograph (m) and magnified view (n) after PA encapsulation. The inset shows a PA-encapsulated microneedle body. **o,p,** Photograph (o) and magnified view (p) after electrodeposition of AuNPs.

backward-facing barbs, as compared to the higher modulus of PDMS that distorts the transferred patterns (Supplementary Fig. 7). The PDMS mould for deformation consists of conical cavities with approximately the same heights as the microneedles and pre-designed angles to define the shape of the deformed barbs (Supplementary Fig. 8a). During compression, the barbs deform to the shape of the cones (Supplementary Fig. 8b). Cones with smaller angles results in more deformed barbs (Supplementary Fig. 8c). This method also allows for the monolithic manufacturing of multiple rows of barbs on a single microneedle (Fig. 3a). Furthermore, a single PDMS mould with individualized cone designs for microneedles of different dimensions allows for the manufacturing of barbs for the entire array in a single process with high yield (Fig. 3b and Supplementary Fig. 9). Tests in ex vivo tissue, using barbs deformed by a 25° mould, indicate that microneedles with backward-facing barbs withstand strong pulling forces (Fig. 3c), which aligns well with simulation results (Fig. 3d and Extended Data Fig. 2b–d). The critical pull-out force, determined by applying different gravitational forces through weights (Extended Data Fig. 2e), increases from unmeasurable, to 18.1, 43.6 and 61.7 mN for microneedles with 0, 1, 2 and 3 rows of barbs, respectively, which also agrees well with simulation results (Extended Data Fig. 2f,g). More accurate insertion tests in rat kidneys, using a high-resolution force gauge, indicate that a bare microneedle easily pulls out of tissue with a critical pull-out force of 0.87 mN (Extended Data Fig. 2h,i and Supplementary Note 2). However, a microneedle with one row of barbs is strongly retained by the tissue and experiences a critical pull-out force of 12.2 mN (Extended Data Fig. 2j,k), which is close to the data from the weight-holding test. Taken together, compared to a bare microneedle (0.87 mN), microneedles with one (12.2 mN–18.1 mN), two (43.6 mN) and three (61.7 mN) rows of barbs achieve enhancement of tissue retention by 14–21, 50 and 71 times, respectively, which are superior to the maximum enhancement of 18 times previously reported^{46,48}. In in vivo tests in rats, bare microneedles are pressed into the kidney but immediately slip out upon removal of the finger due to the restoration force of tissue (Fig. 3e). The impedance of the bare microneedles decreases from 2.5×10^4 k Ω to 2 k Ω upon being pressed into tissue but returns to 2.2×10^4 k Ω upon releasing the finger (Fig. 3f and Supplementary Fig. 10a, $n = 3$ rats). In contrast, barbed microneedles stay in the kidney even with an external pulling force (Fig. 3g, left). Micro-CT imaging of the kidney 7 days post implantation shows that the barbed microneedles remain stably inserted into the kidney, with an insertion depth of ~1 mm (Fig. 3g, right). The impedance of the barbed microneedles decreases from 4.2×10^4 k Ω to 3 k Ω upon being pressed into tissue and stays at ~3 k Ω for at least 7 days in vivo (Fig. 3h and Supplementary Fig. 10b, $n = 3$ rats). Micro-CT images of euthanized rats subject to intense controlled movement with accelerations of up to 40 m s⁻² and angular velocities of up to 750 deg s⁻¹ for 1 h, exceeding the ranges of those during voluntary movements of rats and humans^{70,71}, reveal no noticeable displacement or detachment of the microneedle array from the kidney (Supplementary Fig. 11). All these results collectively suggest the strong tissue retention of the barbed microneedle array that supports stable in vivo recordings.

The implant features an electrically programmable self-destruction mechanism that initiates device degradation upon completion of monitoring. This is based on oxidative corrosion of the external electrode coatings of the microneedles, that is, Au (WE), Pt (CE) and Ag/AgCl (RE), under overpotentials. The Au electrode works stably during electrochemical measurements, showing no sign of degradation in the Au coating after 12 h of amperometric measurement at 0.25 V (Fig. 3i). However, an overpotential exceeding 1.95 V triggers oxidative corrosion of the Au and the underlying Mo⁷². Subsequent mechanical crevices and exfoliations accelerate the disintegration of the Au/Mo coatings. Consistent with literature⁷², the Au/Mo coatings disintegrate within 30 s at 2.5 V, exposing the internal PLGA microneedle (Fig. 3i). Similarly, the Pt (Fig. 3j) and Ag/AgCl (Fig. 3k) electrodes both

disintegrate within 30 s at 2.5 V. Previous studies found no noticeable change in temperature during this process⁷², indicating low possibility of tissue damage from thermal effects. Haematoxylin and eosin (H&E) staining images acquired 24 h after electrode disintegration suggest no noticeable tissue damage (Supplementary Fig. 12).

In vivo tests of the implant in $n = 4$ rats demonstrate the self-destruction process. The microneedles remain visually intact for 1 week in the body (Fig. 3l, left and middle left). At the end of week 1, triggering the dissolution of the metal coatings in vivo exposes the PLGA microneedles (Fig. 3l, middle), which nearly fully bioresorb in the body by the end of week 3 (Fig. 3l, middle right and right) through a US Food and Drug Administration (FDA)-approved, biologically safe hydrolysis process⁷³. The degradation of the entire microneedle array follows a similar pace. It remains visually intact for 1 week in the body (Fig. 3m, left and middle left). Following triggering at the end of week 1, the microneedle array undergoes gradual bioresorption in the body in the subsequent 2–3 weeks (Fig. 3m, middle to right), which also matches the observation from micro-CT imaging (Fig. 1b). Similarly, the e-suture is visually intact by the end of week 1 (Fig. 3n, left and middle left), but shows signs of breakage by the end of week 2 (Fig. 3n, middle) and entirely bioresorbs by week 4 (Fig. 3n, middle right and right). These data collectively suggest that the device is able to support stable in vivo operation for at least 1 week and degrade almost entirely in the subsequent 3 weeks upon triggering self-destruction, matching the need for organ monitoring during the critical first postoperative week and ensuring timely clearance from the body.

Microneedle functionalization for electrochemical sensing

The reported fabrication process offers high-yield and highly reproducible manufacturing of microneedle sensors, as demonstrated by consistent amperometric responses across a 6×6 array in glucose sensing (Supplementary Fig. 13). The microneedle tips are individually coated to yield different sensors. Here we focus on a panel of biochemical markers, including GLU, LA, UA, O₂, K⁺, Na⁺ and pH, that are key indicators of common perioperative organ disorders, such as ischaemia, hypoxia, metabolic suppression, cellular damage, fluid imbalance and inflammation (Supplementary Table 1). For potentiometric sensing of ions (K⁺, Na⁺ and H⁺), sequential coating of a PEDOT:PSS ion-to-electron transducing layer, an ion-selective membrane (ISM) and a polyurethane (PU) antibiofouling layer on AuNP-coated microneedles produces the working electrodes (Fig. 4a). The ISMs selectively allow for the penetration of target ions, thereby creating an ion concentration gradient across the membrane and generating a transmembrane potential indicative of the ion concentration. The K⁺ sensor responds instantly to changes in K⁺ concentration (Supplementary Fig. 14a), as measured by pipetting out old analyte solution and pipetting in new analyte solution while continuously recording sensor response. The transmembrane potential increases linearly as the K⁺ concentration steps up from 1 mM to 32 mM and subsequently returns to similar values as the concentration steps down from 32 mM to 1 mM, indicating a sensitivity of 69.8 ± 2.63 mV per (lg[K⁺]), a good linearity of $R^2 = 0.996$ and good reversibility of sensing (Fig. 4b). Similarly, in the case of Na⁺ sensing (Extended Data Fig. 3a), the transmembrane potential of the Na⁺ sensor changes linearly and reversibly as the Na⁺ concentration cycles from 5 mM to 160 mM and then from 160 mM to 5 mM, exhibiting a sensitivity of 61.04 ± 0.16 mV per (lg[Na⁺]) and a good linearity of $R^2 = 0.999$. For pH sensing (Extended Data Fig. 3b), the transmembrane potential of the pH sensor changes linearly and reversibly as the pH cycles from 4 to 8 and then from 8 to 4, exhibiting a sensitivity of -26.8 ± 1.2 mV per (–lg[H⁺]) and a good linearity of $R^2 = 0.994$. The K⁺, Na⁺ and pH sensors show good sensor-to-sensor reproducibility, with a mean coefficient of variation of 3.8%, 5.3% and 22.8% (Extended Data Fig. 4a–c), respectively. Furthermore, the continuous addition of interfering substances, such as NaCl, KCl, CaCl₂, MgCl₂ and NH₄Cl, minimally affects the transmembrane potential (Supplementary Fig. 15a,b),

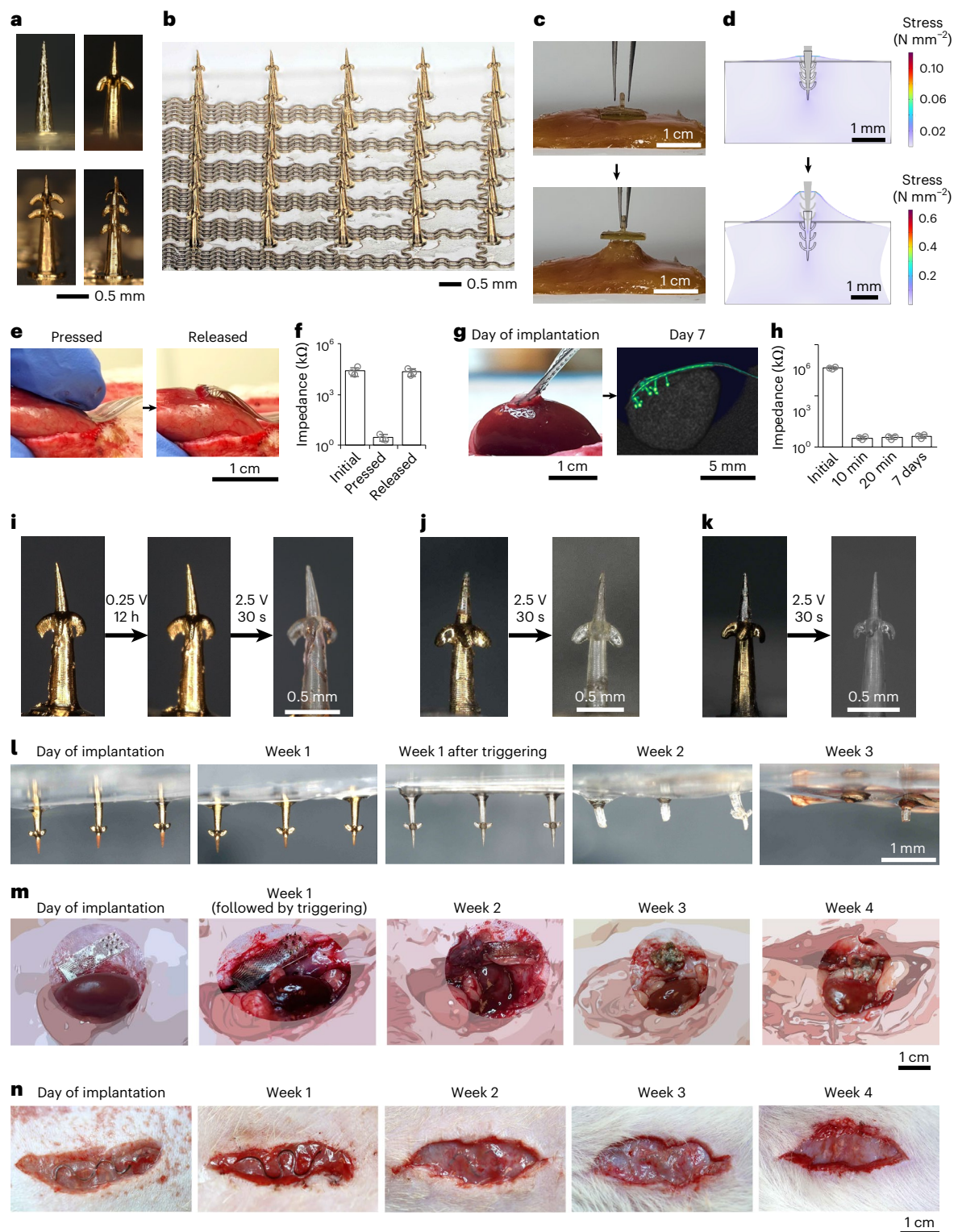


Fig. 3 | In vitro and in vivo characterizations of the implant. **a**, Micrographs of barbed microneedles with different numbers of rows of barbs. The experiments were repeated 3 times independently with similar results. **b**, Magnified view of a 6 × 6 microneedle array with barbs. **c**, Experimental images showing enhanced adhesion of barbed microneedles to a chicken breast sample. **d**, Numerical simulation results showing enhanced adhesion of barbed microneedles to tissue. **e**, Pressing of a bare microneedle array into a rat kidney, which slips out upon releasing. **f**, Impedance of the bare microneedle array at different stages. Data are presented as mean ± s.d. (n = 3 independent experiments). **g**, Stable

affixation of a barbed microneedle array to a rat kidney, which stays in position 7 days after implantation (micro-CT). **h**, Impedance of the barbed microneedle array at different stages. Data are presented as mean ± s.d. (n = 3 independent experiments). **i**, Electrically triggered dissolution of the gold working electrode under an overpotential, with no noticeable change under normal electrochemical measurement. **j**, Electrically triggered dissolution of the Pt counter electrode. **k**, Electrically triggered dissolution of the Ag/AgCl reference electrode. **l**, In vivo degradation of the microneedles. **m**, In vivo degradation of the microneedle array. **n**, In vivo degradation of the e-suture.

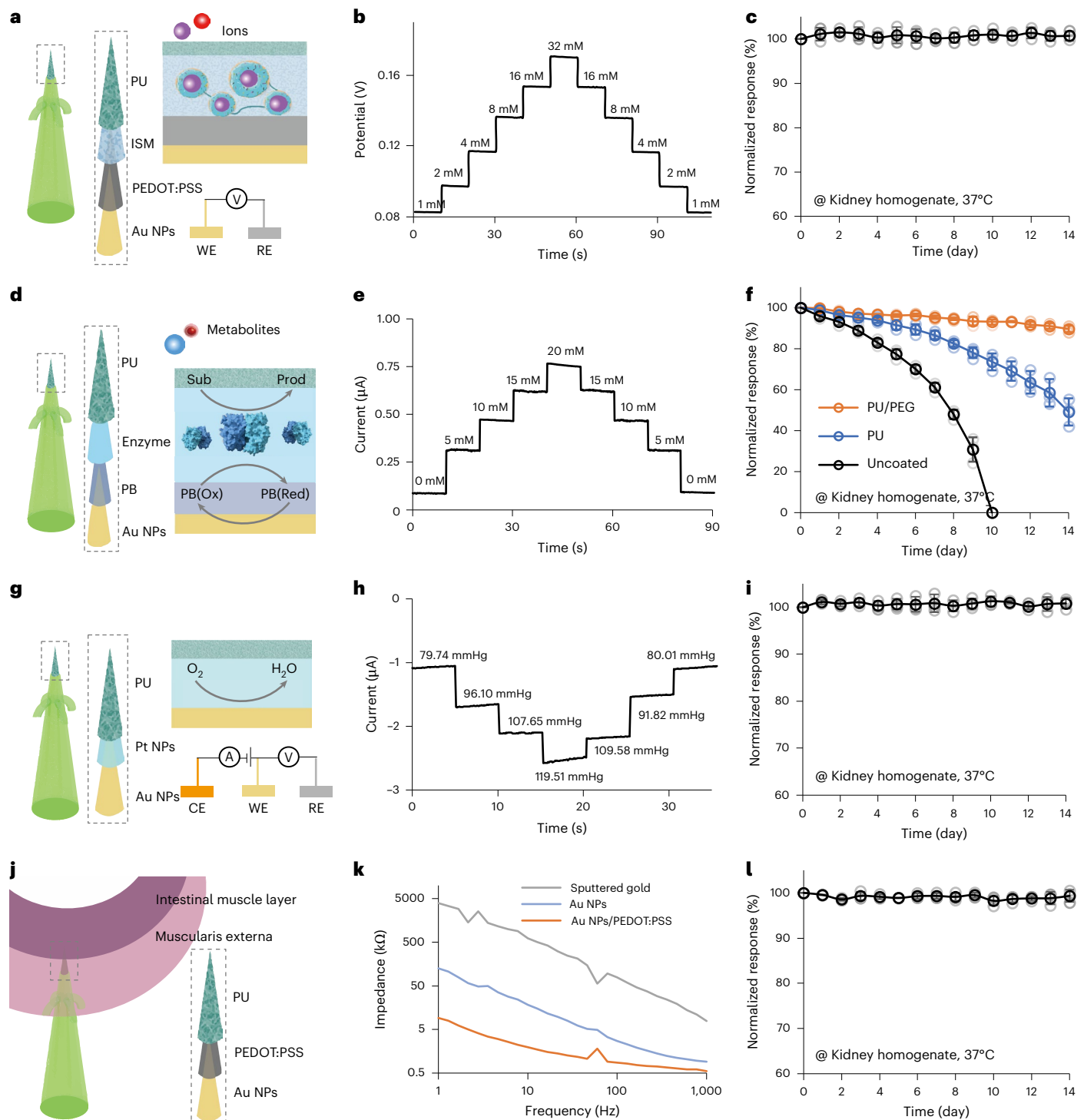


Fig. 4 | Characterizations of the microneedle sensors. a, Schematic illustration of potentiometric sensors. **b**, Potentiometric response of the K⁺ sensor to different concentrations of K⁺. **c**, Chronic stability of the K⁺ sensor in kidney homogenate at 37 °C. Data are presented as mean ± s.d. (*n* = 3 independent sensors). **d**, Schematic illustration of enzyme-based amperometric sensors. **e**, Amperometric response of the glucose microneedle sensor to different concentrations of glucose. **f**, Chronic stability of the glucose sensor with different coatings in kidney homogenate at 37 °C. Data are presented as mean ± s.d. (*n* = 3

independent sensors). **g**, Schematic illustration of the Clark-type oxygen sensor. **h**, Amperometric response of the oxygen sensor to different concentrations of dissolved oxygen. **i**, Chronic stability of the oxygen sensor in kidney homogenate at 37 °C. Data are presented as mean ± s.d. (*n* = 3 independent sensors). **j**, Schematic illustration of the biopotential sensor. **k**, Electrochemical impedance spectroscopy of the biopotential sensor with different coatings. **l**, Chronic stability of the biopotential sensor in kidney homogenate at 37 °C. Data are presented as mean ± s.d. (*n* = 3 independent sensors).

in contrast to the sensitive responses to target ions. Validation of the K⁺, Na⁺ and pH sensors using serum samples indicates respective mean relative errors of 12.7%, 18.4% and 13.0% with respect to the standard mass spectrometry approach (Extended Data Fig. 5a–f)⁷⁴. Chronic

testing of a representative ISM-based ionic sensor (K⁺), combined with a PU-coated microneedle RE, in rat kidney homogenate at 37 °C shows that the normalized sensor response remains stable between 100% and 102% over a period of 2 weeks (Fig. 4c). These results collectively

indicate the good sensitivity, specificity, reproducibility and stability of the microneedle sensors for ion detection.

The monitoring of metabolic biomarkers, including GLU, LA and UA, leverages enzyme-based amperometric sensors in a three-electrode electrochemical system (Fig. 4d). The electrons from enzyme-catalysed oxidation of metabolites, efficiently transferred to the working electrode by electron mediators, generate an electrical current proportional to metabolite concentration. However, the low amplitude of the current resulting from the small sensing area of the microneedle tips limits the detection sensitivity and linear range. To address this limitation, PU serves as a diffusion-limiting layer to broaden the linear range of detection and as an anti-biofouling layer to isolate interfering chemicals⁷⁵. At the same time, Prussian blue (PB) serves as an electron mediator to lower the overpotential applied to the electrode, thereby minimizing interference from other electroactive species, such as UA or ascorbic acid⁷⁶. The GLU sensor responds within seconds upon the addition of analyte of different concentrations (Supplementary Fig. 14b). Under a bias potential of 0.25 V, the microneedle glucose sensor detects GLU in the physiologically relevant range of 0 mM to 20 mM reversibly (Fig. 4e and Supplementary Fig. 16a), with a sensitivity of $0.087 \pm 0.012 \mu\text{A mM}^{-1}$, a good linearity of $R^2 = 0.9917$ and a mean coefficient of variation of 9.9% across three samples (Extended Data Fig. 4d). In contrast to glucose-induced signals, sequential addition of 0.5 mM UA, 0.5 mM LA, 10 mM KCl, 0.01 mM vitamin C (VC), 1 mM NaCl, 0.5 mM UA, 0.5 mM LA, 10 mM KCl, 10 mM NaCl and 0.5 mM CaCl_2 , generates negligible currents (Supplementary Fig. 15c). Similarly, LA oxidase-coated and UA oxidase-coated microneedles serve as the sensors for LA and UA, respectively. Under a bias potential of -0.35 V , the current changes reversibly with LA concentration in the range of 0 mM–20 mM (Extended Data Fig. 3c and Supplementary Fig. 16b), with a sensitivity of $0.025 \pm 0.001 \mu\text{A mM}^{-1}$, a good linearity of $R^2 = 0.9995$ and a mean coefficient of variation of 13.0% across three samples (Extended Data Fig. 4e). Under a bias potential of 0.25 V, the current changes reversibly with UA concentration in the range of 0–1 mM (Extended Data Fig. 3d and Supplementary Fig. 16c), with a sensitivity of $0.13 \pm 0.005 \mu\text{A mM}^{-1}$, a good linearity of $R^2 = 0.9948$ and a mean coefficient of variation of 11.3% across three samples (Extended Data Fig. 4f). Similar interference tests show that interfering chemicals generate negligible signals on the LA and UA sensors (Supplementary Fig. 15d,e), suggesting good selectivity of sensing. Validation of the GLU, LA and UA sensors using kidney homogenate samples indicates relative errors of 8.0%, 11.7% and 10.0% with respect to commercial, medical-grade GLU, LA and UA metres (Extended Data Fig. 5g–i), respectively⁷⁷. Chronic testing of a representative enzymatic sensor (GLU), combined with a PU-coated microneedle RE and CE, in rat kidney homogenate at 37°C shows that uncoated GLU sensors gradually lose activity by day 10 and PU-coated GLU sensors preserve 49.0% current response by day 14 (Fig. 4f). However, the use of a polyethylene glycol (PEG) modified PU layer maintains 89.6% current response by day 14, thereby enabling chronic monitoring with minimal drift in sensor response (Fig. 4f).

The measurement of tissue oxygenation, also known as oxygen partial pressure (PO_2), utilizes the gold-standard Clark method⁷⁸. Electrodeposited Pt NPs on AuNP-coated microneedles catalyses the reduction of oxygen to generate an electrical current proportional to dissolved oxygen concentration (Fig. 4g). It responds instantly to changes in oxygen concentration (Supplementary Fig. 14c). Under a bias voltage of -0.65 V , the amplitude of the current signal changes reversibly with the concentration of oxygen in the PBS (Fig. 4h and Supplementary Fig. 15d), with a sensitivity of $-0.037 \pm 0.006 \mu\text{A mmHg}^{-1}$ and a good linearity of $R^2 = 0.9995$. It shows good sensor-to-sensor reproducibility, with a mean coefficient of variation of 2.1% (Extended Data Fig. 4g). Chronic testing of the oxygenation sensor in rat kidney homogenate at 37°C shows that the normalized

sensor response remains stable over a period of 2 weeks (Fig. 4i). High-fidelity recording of biopotential critically relies on low impedance electrodes⁷⁹. This work leverages a tri-layer coating of Au/AuNPs/PEDOT:PSS on microneedles to faithfully record organ electrophysiology (Fig. 4j). Electrochemical impedance spectroscopy (EIS) reveals electrical impedance of $7.8 \times 10^5 \Omega$, $2.7 \times 10^3 \Omega$ and 875Ω at 100 Hz for microneedles with Au, Au/AuNPs and Au/AuNPs/PEDOT:PSS coatings, respectively, highlighting nearly 3 orders of magnitude improvement by the tri-layer design (Fig. 4k). Benchtop testing suggests that the microneedle electrodes are capable of recording mV-scale sinusoidal signals in PBS (Supplementary Fig. 15e), thereby meeting the needs of electromyography (EMG) recording (typical range: 0–10 mV)⁸⁰ in this work. Chronic testing of the biopotential sensor in rat kidney homogenate at 37°C shows that the normalized sensor response remains stable over a period of 2 weeks (Fig. 4l).

Multimodal monitoring of organ health in rodent models

Demonstrations of multiparametric biochemical monitoring of deep organs involve implanting devices with a 3×3 microneedle sensor array in rodent models (adult Sprague–Dawley rats) using the surgical procedure described in Methods and Supplementary Fig. 17. The first study focuses on organ ischaemia, a common intraoperative complication that may result from low blood pressure, blood vessel injuries and/or unsuccessful blood vessel connection in organ transplantation surgery⁸¹. This condition may also emerge post surgery, such as in cases of organ transplant rejection³². The experiment simulates acute kidney ischaemia through periodic occlusion of the renal artery and the renal vein using haemostatic forceps (Fig. 5a). The microneedles penetrate the renal capsule, which isolates and insulates the microenvironment of the kidney, and probe the renal cortex to provide accurate monitoring of renal metabolism. In this demonstration, the device monitors renal oxygenation (PO_2), LA and UA (Fig. 5b, $n = 3$ rats). As the kidney undergoes 15-min cycles of ischaemia and recovery, the device detects prompt decreases in PO_2 to $23.3 \pm 8.6 \text{ mmHg}$ upon ischaemia and resumption of PO_2 to a baseline of $38\text{--}40 \text{ mmHg}$ upon restoration of blood supply. Concurrent monitoring of renal LA and UA during these cycles, however, reveals steady increases in LA from $0.85 \pm 0.8 \text{ mM}$ to $9.9 \pm 3.3 \text{ mM}$ and UA from $0.06 \pm 0.02 \text{ mM}$ to $0.45 \pm 0.11 \text{ mM}$ over the course of 80 min. These findings might result from irreversible organ injury due to ischaemia or reperfusion organ injury upon restoration of blood supply due to oxidative stress and inflammation⁸². The differential responses of different biomarkers to ischaemia highlight the need for concurrent monitoring of multiple biomarkers, including oxygenation and biochemical markers, to ensure comprehensive assessment of organ health. To demonstrate the capability of spatiotemporally resolved monitoring of organ states, a 3×3 device maps renal oxygenation over 3×3 sites in the renal cortex during ischaemia–recovery cycles (Fig. 5c). The device reveals spatial heterogeneity of oxygenation across different areas of the renal cortex and its temporal evolution upon the induction of hypoxia and recovery from hypoxia, showcasing capability in high spatiotemporal resolution monitoring of organs.

The second study focuses on a chronic kidney ischaemia model to demonstrate the capability of the device for biochemical monitoring of deep organs for at least 1 week post operation. Occlusion of the blood vessels of the left kidney by surgical suture followed by implantation of the device establishes the animal model. The device monitors the glucose concentration in the ischaemic kidney for 7 days, during which intraperitoneal injections of glucose serve as an approach to modulate renal glucose levels. A commercial glucose metre provides reference blood glucose levels. In the healthy control (Fig. 5e, $n = 3$ sensors), the sensors reliably track fluctuations in renal glucose levels following glucose injections, demonstrating a strong correlation with blood glucose levels across all three measurement days (days 0, 3 and 7). In the ischaemia group (Fig. 5f, $n = 3$ sensors), the sensors

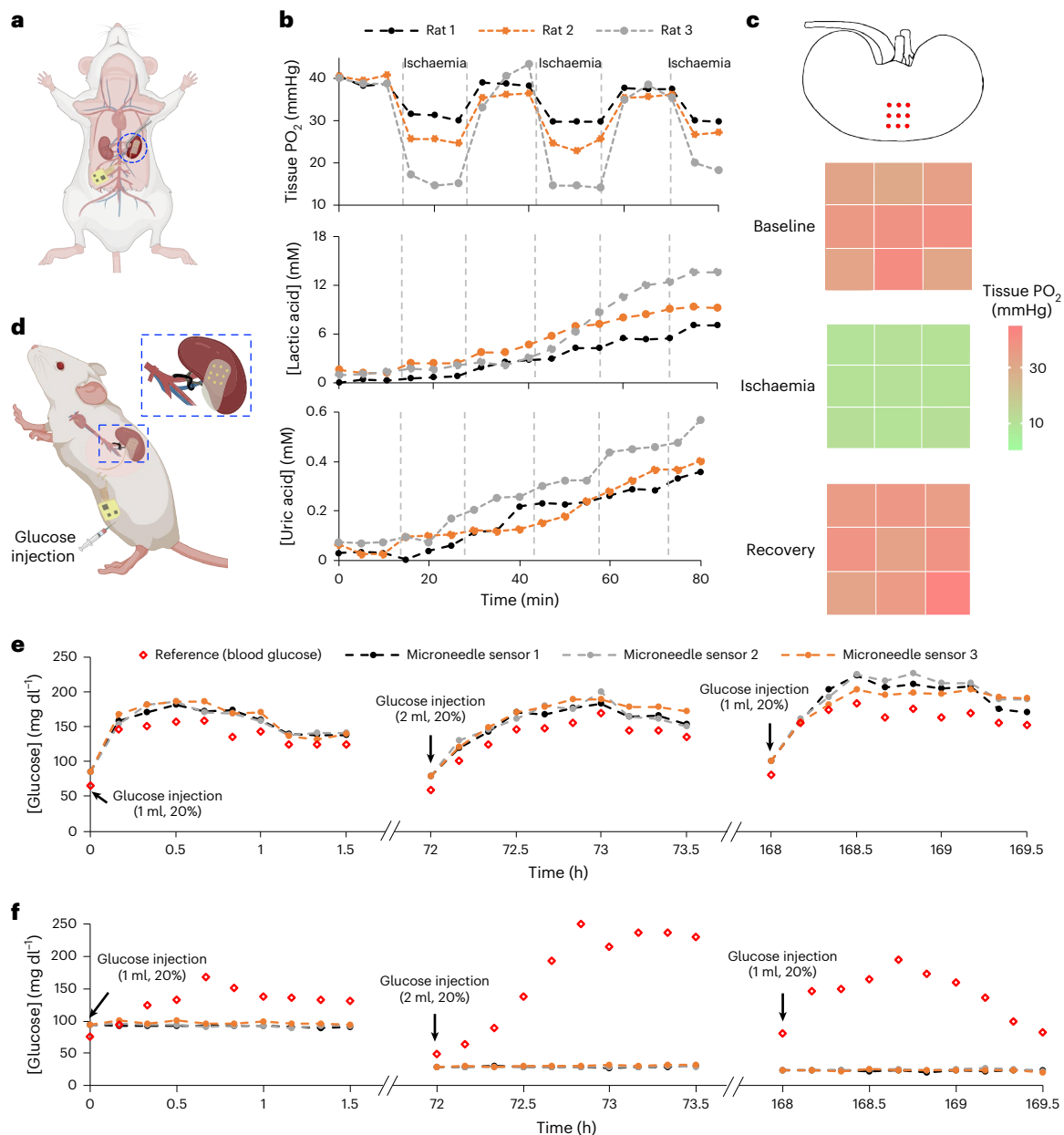


Fig. 5 | Monitoring of acute and chronic kidney ischaemia in rats. a, Schematic illustration of acute kidney ischaemia monitoring. Created with BioRender (<https://BioRender.com/82rlsyn>). **b**, Concurrent monitoring of kidney oxygenation, LA and UA under ischaemia cycles ($n = 3$ rats). **c**, Spatiotemporal mapping of kidney oxygenation under ischaemia cycles ($n = 3$ rats). **d**, Schematic

illustration of chronic kidney ischaemia monitoring. Created with BioRender (<https://BioRender.com/o3rie0t>). **e**, Chronic monitoring of renal glucose levels in the healthy control ($n = 3$ sensors). **f**, Chronic monitoring of renal glucose levels in the ischaemia model ($n = 3$ sensors).

detect minimal changes in renal glucose levels despite fluctuations of blood glucose levels following glucose injections, consistent with blocked blood supply to the kidney. The sensors also capture progressive hypoglycaemia, with glucose levels decreasing from ~ 100 mg dl^{-1} on day 0 to ~ 28 mg dl^{-1} on day 3 and ~ 23 mg dl^{-1} on day 7, further demonstrating their capability in chronic glucose monitoring.

The final set of studies investigate postoperative gut disorders, such as nutrient absorption disorders, gut dysmotility and anastomotic leaks, that may arise from gastrointestinal surgery, anaesthesia and medications, and/or systemic inflammation and infection⁸³. The device is capable of monitoring nutrient absorption within the lumen, tracking gut motility through EMG measurements on the intestinal wall and detecting anastomotic leaks into the abdominal cavity, by adjusting the lengths of the microneedles. The device forms a cuff

around a segment of the small intestine (jejunum), with the microneedles minimally invasively probing the lumen or the intestinal wall (Extended Data Fig. 6a,b). Direct injection of electrolytes (PBS) and a nutrient (glucose) into the small intestine of anaesthetized rats simulates food intake in this experiment. Upon injection, the device detects abrupt increases in the levels of glucose, Na^+ , K^+ and pH that match those in the solution and their gradual decreases during absorption (Extended Data Fig. 6c). Specifically, the glucose concentration in the lumen decreases from 20.0 ± 1.0 to 11.6 ± 3.2 mM within 1 h. Simultaneously, the Na^+ concentration changes from 187.5 ± 17.7 mM to 190.3 ± 2.8 mM and the K^+ concentration decreases from 7.6 ± 0.7 mM to 4.8 ± 0.8 mM. The pH decreases from ~ 7.1 to 6.4 ± 0.2 during this process. These results highlight the device's capability in monitoring the gut's absorption function. A separate set of

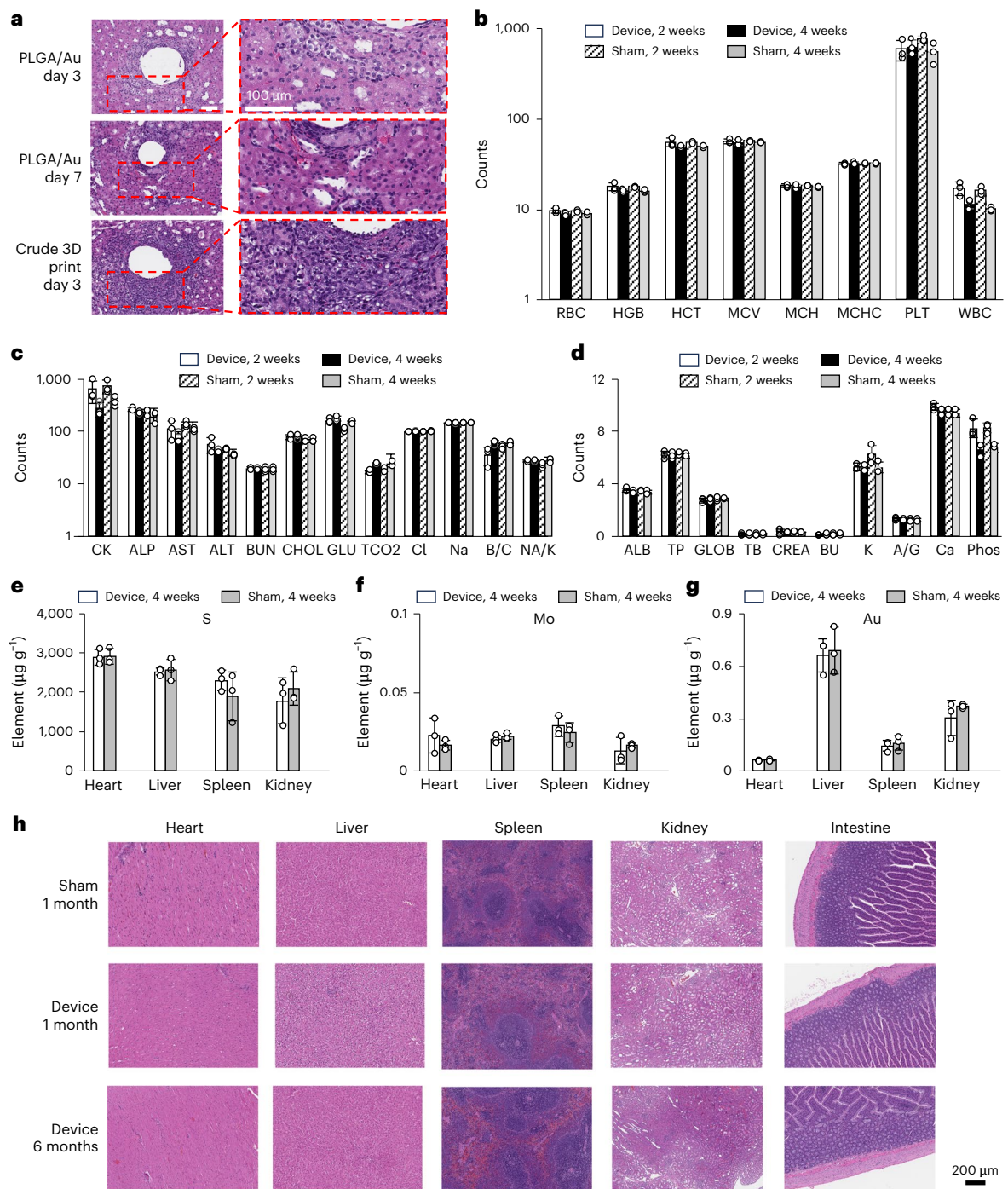


Fig. 6 | Biocompatibility study of the implant. **a**, H&E staining images of the tissue at the insertion sites of Au-coated microneedles (days 3 and 7) and crude 3D-printed microneedles (day 3). **b**, Complete blood counts of device and sham groups ($n = 3$ rats per group) at 2 and 4 weeks. Analysis of complete blood counts. RBC, red blood cells ($\times 1,000,000 \mu\text{l}^{-1}$); HGB, blood haemoglobin level (g dl^{-1}); HCT, haematocrit level (%); MCV, mean corpuscular volume (fl); MCH, mean corpuscular haemoglobin (pg); MCHC, mean corpuscular haemoglobin concentration (g dl^{-1}); PLT, platelet count in blood ($\times 1,000 \mu\text{l}^{-1}$); WBC, white blood cells ($\times 1,000 \mu\text{l}^{-1}$). **c**, Blood chemistry tests of device and sham groups ($n = 3$ rats per group) at 2 and 4 weeks. Analysis of complete blood chemistry. CK, creatine kinase (U l^{-1}); ALP, alkaline phosphatase (U l^{-1}); AST, aspartate transaminase (U l^{-1}); BUN, blood urea nitrogen (mg dl^{-1}); CHOL, cholesterol

(mg dl^{-1}); GLU, glucose (mg dl^{-1}); TCO₂, bicarbonate (mmol l^{-1}); Cl, chloride (mmol l^{-1}); Na, sodium (mmol l^{-1}); B/C, blood urea nitrogen/creatinine ratio; NA/K, sodium/potassium ratio. **d**, Blood chemistry tests of device and sham groups ($n = 3$ rats per group) at 2 and 4 weeks. Analysis of complete blood chemistry. ALB, albumin (g dl^{-1}); TP, total protein (g dl^{-1}); GLOB, globulin (g dl^{-1}); TB, total bilirubin (mg dl^{-1}); CREA, creatinine (mg dl^{-1}); BU, bilirubin unconjugated (mg dl^{-1}); K, potassium (mmol l^{-1}); A/G, albumin/globulin; Ca, calcium (mg dl^{-1}); Phos, phosphorus (mg dl^{-1}). **e–g**, Sulfur (**e**), Mo (**f**) and Au (**g**) levels in the heart, liver, spleen and kidney of device and sham groups ($n = 3$ rats per group) at 4 weeks. **h**, Representative images of H&E-stained tissue sections of the heart, liver, spleen, kidney and intestine of sham and device groups at 1 and 6 months ($n = 3$ rats per group). All data are presented as mean $\pm$ d.

experiments investigate (Extended Data Fig. 6d) gut motility under different stimulations in anaesthetized rats by using microneedles inserted into the intestinal wall without reaching the lumen. Following the initial injection of PBS, the gut does not exhibit noticeable myoelectric activities within a 5-min window. However, subsequent injection of PBS/glucose (20 mM) elicits noticeable myoelectric activities within 5 min. In the second group receiving PBS/capsaicin (100 μ M) injection after PBS injection, the gut also exhibits more substantial myoelectric activities within 5 min. These findings demonstrate the device's potential in assessing gut dysmotility.

Biocompatibility

Complete devices are implanted in rats, triggered at week 1 and degrade in the body over the subsequent 3 weeks. Throughout this period, various tests, including tissue histology, blood tests, trace element analysis and weight tracking, assess the biocompatibility and health effects of the device. Au-coated PLGA microneedles elicit minimal inflammation in rat kidneys on Days 3 and 7 ($n = 3$ rats), as shown by H&E staining (Fig. 6a (top and middle) and Supplementary Fig. 18a,b), consistent with findings from other microneedle devices⁷². In contrast, crude 3D-printed microneedles induce strong infiltration of inflammatory cells even by day 3 (Fig. 6a (bottom) and Supplementary Fig. 18a,b). Complete blood counts and blood chemistry tests reveal no significant differences between sham and device groups at 2 and 4 weeks ($n = 3$ rats per group, $P < 0.05$, one-way analysis of variance (ANOVA)) across blood cell populations, electrolytes, enzymes and metabolites (Fig. 6b–d). These data suggest no device-related systemic toxicity or disruption of physiology. Trace element analyses by mass spectrometry confirm no abnormal accumulation of dissolved Au, Mo and S in organ tissues (heart, liver, spleen and kidney) compared to sham at 4-week endpoints ($n = 3$ rats per group). The total masses of Ag (28 ng) and Pt (11 ng) in the device (Supplementary Fig. 19) only constitute a trace fraction of allowable doses in the body⁸⁴. In separate cohorts, weekly tracked body weights up to 5 weeks and endpoint organ weights at 5 weeks reveal no significant differences ($P < 0.05$, one-way ANOVA) between sham and device groups ($n = 3$ rats per group) (Supplementary Fig. 18c,d), indicating normal growth of implanted animals. Furthermore, H&E staining images of the heart, liver, spleen, kidney and intestine reveal no noticeable differences between the sham group, device group at 1 month and device group at 6 months ($n = 3$ rats per group), indicating no long-term toxicity. All these data collectively indicate the biocompatibility of the bioresorbable device for chronic monitoring of deep organs.

Discussion

This study presents an integrated bioresorbable microneedle-based system for comprehensive, continuous and chronic biochemical monitoring of deep organ function throughout the perioperative period. The device enables multiplexed, real-time and spatially mapped electrochemical sensing while addressing key challenges in deep organ interfacing, stable affixation, chronic functionality and controlled bioresorption. By leveraging a 3D printing-based, photolithography-free fabrication approach, the microneedle array achieves conformal organ interfacing, 3D probing of organ parenchyma and stable retention through backward-facing barbs, allowing for high-fidelity data acquisition with minimal irritation. The system further incorporates a bioresorbable, rolling-based e-suture for multiplexed electrical interconnection while preventing displacement, eliminating the need for secondary surgery for retrieval. The electrically programmable self-destruction mechanism ensures controllable bioresorption of the implant, mitigating risks associated with long-term foreign body retention. Demonstrations in rodent models validate the system's ability to detect and track clinically relevant complications, such as ischaemia, metabolic disturbances and gastrointestinal dysfunction. The ability to perform continuous biochemical monitoring

in deep organs represents an advancement over conventional intraoperative vital sign tracking and intermittent blood tests, which often fail to detect localized pathology in a timely manner. A limitation of this work is the use of non-bioresorbable biochemical sensing coatings and protective layers, although they constitute only a trace fraction of the total mass. These include ISMs, PU and PEDOT:PSS, all of which have bioresorbable forms or replacements^{85–87}. Future work would involve incorporating these materials to enhance full-system degradability. Another direction of interest would be developing advanced electrochemical analysis techniques on microneedles to enable the monitoring of cytokines, drugs and endotoxin, thereby opening up possibilities for investigating inflammation, infection and drug metabolism in organs. In addition, further refinement of anti-biofouling coatings and encapsulation materials could extend the monitoring period to months to enhance long-term functionality. Finally, while the current device represents a research-stage prototype, many of its key manufacturing steps, such as injection moulding, metal deposition, and the coating of enzymes and protective layers (as used in continuous glucose monitoring devices), are already industrially established and commercially implemented. Future work would involve further optimization of the process to facilitate clinical translation.

Methods

Electrical components

The system was assembled on a double-layer fPCB [Cu (18 μ m): Polyimide (PI, 75 μ m): Cu (18 μ m)]. A low-dropout linear voltage regulator (ADP7112, Analog Devices) in a 6-pin wafer-level chip-scale package (WLCSP) (1.2 mm $\times$ 1 mm) converted the DC voltage to a constant voltage supply (3.3 V) to power the system. A BLE SoC (nRF52832-CIAA, Nordic Semiconductor) in WLCSP packaging (3.0 mm $\times$ 3.2 mm) served as the CPU and Bluetooth communication module. The BLE SoC used a miniaturized (3.2 mm $\times$ 1.6 mm) ceramic 2.45 GHz antenna (2450AT18A100, Johanson Technology) for wireless communication. The potentiometric and amperometric circuitry consists of a voltage buffer array (ADA4505, Analog Devices), a 32-channel switch array (ADG732, Analog Devices) and an electrochemical analogue front end (AD5941, Analog Devices). The use of other passive components with 0201 (imperial) packaging minimized the overall size of the system.

Fabrication of the implant

The microneedle array design referenced that described in a previous publication⁴³. The design was exported to a 3D printer (ELEGOO Saturn 3 Ultra) and printed at a resolution of 10 μ m per layer with an exposure time of 7 s. Barbed microneedle structures were formed by inserting the printed array into a PDMS mould containing cone-shaped cavities, which mechanically deformed the horizontal barbs. The array was clamped using binder clips and cured under 405 nm UV light for 3 min, after which the PDMS mould was carefully removed. Uncured Ecoflex (00-31) was then cast over the 3D-printed structure. After degassing under vacuum and curing at 70 °C for 3 h, the 3D-printed resin was removed, resulting in a negative Ecoflex mould containing both barbed microneedle and serpentine patterns. This mould was temporarily bonded to a glass slide to form a sealed chamber and filled with PLGA in a vacuum oven at 160 °C. After cooling, the mould was removed, yielding a PLGA microneedle array with integrated barbed structures and serpentine interconnects on a glass substrate. To create conductive features, 500 nm of molybdenum (Mo) was sputtered over the entire structure. A water-soluble polyvinyl alcohol (PVA) tape, patterned with a CO₂ laser to match the microneedle positions, was aligned and applied to selectively expose the microneedles. Then, 100 nm of gold was sputtered only onto the exposed microneedles. Removing the PVA tape transferred the microneedle array and serpentine traces from the glass slide onto the tape. The PVA tape carrying the microneedle array was flipped to expose the backside and plasma treated (Harrick Plasma, PDC001HP) for 5 min. A 20- μ m-thick PLGA film

was surface treated using a corona treater (BD-20AC, ETP) for 1 min, then laminated with the treated microneedle array. The PVA tape was dissolved in water, and the structure was dried using an air gun. An additional 20- μm -thick PLGA film was patterned with microneedle-aligned holes using a CO_2 laser, overlaid onto the microneedle-transferred film and thermally bonded on a hot plate at 80 °C for 4 h to form a unified structure. Finally, to create the e-suture, the integrated microneedle device was rolled around a thin wire as a central axis on a hot plate at 80 °C to form a coiled interconnect. The distal segment was left unheated to allow unrolling post implantation for connection to external electronics.

Device encapsulation and integration

Preparation of the PA was conducted according to the literature⁵³. Component 1 consisted of 4-pentenoic anhydride, 1,3,5-triallyl-1,3,5-triazine-2,4,6(1H,3H,5H)-trione (TTT) and 1,4-butanedithiol (BDT) mixed at a molar ratio of 1:4:7. Component 2 included poly(ethylene glycol) diacrylate with a number average molecular weight M_n of 575, 4-pentenoic anhydride and pentaerythritol tetrakis(3-mercaptopropionate) (PETMP) mixed at a mass ratio of 1:0.73:1.73. Components 1 and 2 were then combined at a 1:1 volume ratio and thoroughly mixed. To this mixture, 30 mg of 2,2-dimethoxy-2-phenylacetophenone (DMPA) was added per 1 ml as a photoinitiator. The resulting formulation was poured into a mould containing the microneedle array and UV cured at 365 nm for 5 min to form a 100- μm -thick PA encapsulation layer. A PDMS slab was then applied to screen the microneedle tips, self-stopping at the barbs. This was followed by spin coating at 2,000 r.p.m. to deposit an additional -10- μm -thick PA layer over the microneedle body. Before encapsulation, water-soluble tape was used to protect the electrical interconnects at the base, allowing for easy connection to the electronics module afterwards.

Preparation of the Au/AuNP microneedle electrode

Au NPs were deposited on the tips of the microneedles after a 400-s plating reaction in a buffer solution containing 0.2 mM HAuCl_4 (Sigma-Aldrich), 0.01 M HCl (Sigma-Aldrich) and 2 mM sodium citrate (Sigma-Aldrich) at a current of -1 mA. Following drying, the microneedle tip was immersed in a PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). Finally, air drying of the electrode for 12 h at ambient temperature completed the fabrication process.

Preparation of the Au/AuNP/PtNP microneedle electrode

The Au/AuNP microneedle electrode was placed in a platinum-plating solution consisting of chloroplatinic acid (8 mM H_2PtCl_6 and 50 mM HCl) (Sigma-Aldrich). Pt NPs were deposited onto the Au/AuNP microneedle electrode after 200 s of electroplating reaction at a current of -1 mA. Following drying, the microneedle tip was immersed in a PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). Finally, air drying of the electrode for 12 h at ambient temperature completed the fabrication process.

Preparation of the Au/AuNP/PB microneedle electrode

An Au/AuNP microneedle electrode was placed in a solution consisting of 0.25 mol l^{-1} potassium chloride (KCl), 0.05 mol l^{-1} anhydrous ferric chloride (FeCl_3) and 0.05 mol l^{-1} potassium ferricyanide ($\text{K}_3[\text{Fe}(\text{CN})_6]$) (Sigma-Aldrich). PB was deposited onto the Au/AuNP microneedle electrode after 120 s of electroplating reaction at a current of -0.5 mA to prepare the Au/AuNP/PB microneedle electrode.

Preparation of the Au/AuNP/PEDOT:PSS microneedle electrode

The Au/AuNP/PEDOT:PSS microneedle electrode was obtained by immersing the Au/AuNP microneedle electrode in 30 ml of a mixture

of 0.01 M methylene dioxythiophene monomer (EDOT) (Sigma-Aldrich) and 0.1 M sodium polystyrene sulfonate (NaPSS) (Sigma-Aldrich) and applying a constant current of 140 μA to the electrode for 200 s. The Au/AuNP/PEDOT:PSS microneedle electrodes were used for EMG recording.

Preparation of the microneedle counter electrode and microneedle reference electrode

The Au/AuNP/PtNP microneedle electrode was used as the counter electrode. The fabrication of the Ag/AgCl reference electrode involved immersing gold-coated microneedle electrodes in Ag evaporation environment to directly coat an Ag layer on the electrode. Subsequently, the electrode was treated in 0.1 M FeCl_3 solution for 5 min. For the preparation of reference electrodes with additional polyvinyl butyral (PVB) coating, 79.1 mg of PVB and 50 mg of sodium chloride were dissolved in 1 ml of methanol. The electrode was then immersed in this solution for 3 h. After removal from the solution, the electrode was treated at 90 °C for 60 min to complete the fabrication of the PVB reference electrode. Following overnight drying, the microneedle PVB reference electrode was immersed in PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). Finally, air drying of the microneedle PVB reference electrode for 12 h at ambient temperature completed the fabrication process.

Preparation of glucose, uric acid, lactic acid and O_2 -sensing microneedle electrode

(1) For glucose detection, a mixture consisting of glucose oxidase (50 mg ml^{-1} , Sigma-Aldrich), bovine serum albumin (BSA, 80 mg ml^{-1} , Sigma-Aldrich) and glutaraldehyde (2.5 wt% in PBS, Sigma-Aldrich) was prepared in PBS at a volume ratio of 1:5:2. The Au/AuNP/PB microneedle electrodes were drop coated three times in the glucose oxidase precursor solution described above. Following overnight drying, the electrodes were immersed in a PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). Finally, air drying of the resultant Au/AuNP/PtNP/GOX microneedle electrodes for 12 h at ambient temperature completed the fabrication process. (2) For uric acid detection, uric acid oxidase (15 mg ml^{-1} , Sigma-Aldrich), BSA (80 mg ml^{-1} , Sigma-Aldrich) and glutaraldehyde (2.5 wt% PBS, Sigma-Aldrich) were combined in PBS at a volume ratio of 1:5:2. The Au/AuNP/PB microneedle electrodes were processed using the dip-coating technique in the uric acid oxidase precursor solution, followed by overnight drying and immersion in PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). Finally, air drying of the resultant Au/AuNP/PB/Uricase microneedle electrodes for 12 h at ambient temperature completed the fabrication process. (3) For lactate sensing, lactate oxidase (30 mg ml^{-1} , Sigma-Aldrich), BSA (80 mg ml^{-1} , Sigma-Aldrich) and glutaraldehyde (2.5 wt% PBS, Sigma-Aldrich) were dissolved in PBS at a volume ratio of 1:5:2. The Au/AuNP/PB microneedle electrodes underwent the dip-coating process in the lactate oxidase solution three times, followed by overnight drying and immersion in PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich). The electrodes, designated as Au/AuNP/PB/LOx microneedles, were subsequently dried for 8 h at room temperature. The preparation of PEG-modified PU was conducted in accordance with the methods described in the literature⁸⁸. First, PU was dissolved in a toluene solution containing 10% (w/v) hexamethylene diisocyanate and 2.5% (w/v) trimethylamine, and stirred at 50 °C for 2.5 h under a nitrogen atmosphere. The resulting mixture was then reacted with a PEG-toluene solution containing 2.5% (w/v) triethylamine at 50 °C for 24 h. Finally, the PU-PEG coating was cast onto the enzyme-based microneedle electrode. (4) The Au/AuNP/PtNP microneedle electrode was used as the O_2 electrode based on the Clark oxygen sensing method⁷⁸. Following overnight drying, the microneedle tip was immersed in a PU solution (0.6 g of PU

dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich).

Preparation of Na⁺, K⁺ and pH-sensing microneedle electrodes

The preparation of the Na⁺-selective membrane cocktail was conducted using a composition that included Na ionophore X (1% (w/w), Sigma-Aldrich), sodium tetrakis[3,5-bis(trifluoromethyl)phenyl]borate (Na-TFPB, 0.55% (w/w), Sigma-Aldrich), polyvinyl chloride (PVC, 33% (w/w), Sigma-Aldrich) and dioctyl sebacate (DOS, 65.45% (w/w), Sigma-Aldrich). A quantity of 200 mg of this cocktail was solubilized in 1,320 µl of tetrahydrofuran (THF) under agitation for 30 min. For the K⁺-selective membrane, the formulation comprised valinomycin (2% (w/w), Sigma-Aldrich), potassium tetrakis[phenyl]borate (NaTPB, 0.5% w/w, Sigma-Aldrich), PVC (32.7% w/w) and DOS (64.8% w/w). Here, 200 mg of the mixture was dissolved in 700 µl of cyclohexanone to fabricate the membrane solution. ISMs for Na⁺ and K⁺ were fabricated through a dip-coating process of the corresponding electrodes into their respective membrane cocktails, followed by air-drying at ambient temperature overnight. The pH-sensing coating was synthesized by dissolving 20 mg of polyaniline emeraldine salt in 20 ml of dimethyl sulfoxide (DMSO, Sigma-Aldrich). To achieve complete dissolution, the mixture was stirred continuously for 24 h. The microneedle electrodes were then coated by dip coating in the polyaniline emeraldine solution and dried for 2 h at 60 °C, thus forming a polyaniline emeraldine base membrane on the gold electrode surface. Subsequently, the membrane was exposed to a 1 ml solution of hydrochloric acid (HCl, 1 mol l⁻¹, Sigma-Aldrich) within a vacuum chamber for 6 h to ensure thorough protonation. Following overnight drying, the microneedle tip was immersed in a PU solution (0.6 g of PU dissolved in 10 g of a 98:2 mass ratio tetrahydrofuran/dimethylformamide solution, Sigma-Aldrich).

Surface morphology characterization of sensing electrodes

The surface morphology of the microneedle sensing electrodes was imaged by a benchtop scanning electron microscope (SEM, Helios 5 CX DualBeam, Thermo Fisher). Micrographs of the samples were taken using an optical microscope (VHX-7000N, Keyence).

Electrochemical characterization of microneedle sensing electrodes

The microneedle electrodes (working electrodes) used for biochemical assays were characterized using an electrochemical workstation (PalmSens 4). The selectivity of the electrochemical sensors for glucose, lactate, uric acid, O₂, sodium ions, potassium ions and pH were evaluated separately by sequentially adding certain concentrations of interfering substances to the solution. For the ion sensors, the electrochemical workstation characterized the open-circuit potential of the two-electrode system, where a microneedle Ag/AgCl electrode was used as the reference electrode during the test. The open-circuit potential was recorded for different concentrations of the substance to be measured. A series of ion solutions with different concentrations was prepared, in which deionized water was used as the solvent. The test was paused for 30 s each time the concentration of the substance changed to allow for sufficient mass transfer in the solution. Interference studies were carried out by adding 10 mM NaCl, 10 mM KCl, 0.5 mM CaCl₂ or 5 mM NH₄Cl dissolved in deionized water. For the metabolite sensors, a three-electrode system (working, counter and reference electrodes) was used, where the amperometric sensors were applied bias potentials of 0.25 V, -0.35 V and 0.25 V for glucose, lactate and uric acid, respectively. A microneedle Au/AuNP/PtNP electrode was used as the counter electrode, and a microneedle Ag/AgCl electrode was used as the reference electrode during the test. Interference studies were performed by adding 1 mM UA, 0.5 mM LA, 0.01 mM VC, 10 mM NaCl, 0.5 mM CaCl₂ or 10 mM KCl dissolved in deionized water. The effect of the material on overall sensor performance was assessed by measuring the electrochemical properties of electrodes with different material

structures. The sensitivity, detection selectivity and detection range of each electrode were analysed separately. For the oxygen sensor, a three-electrode system (working electrode, counter electrode and reference electrode) was used, with a bias potential of -0.65 V applied. A microneedle Au/AuNP/PtNP electrode was used as the counter electrode and a microneedle Ag/AgCl electrode was used as the reference electrode during the tests. Dissolved oxygen concentration in PBS was measured using the Digital Tester Smart Bluetooth dissolved oxygen metre (RMR-6NW-2QU355, Buiishu). The concentration of oxygen in PBS was regulated by continuous passage of air or heating.

Validation of microneedle sensors against standard methods in complex biofluids

Each microneedle sensor was evaluated across four concentration gradients in complex biofluids. For K⁺ sensing, the test conditions included: undiluted serum, twofold-diluted serum, serum spiked with 2 mM KCl (final concentration) and serum spiked with 5 mM KCl (final concentration). For Na⁺ sensing, the conditions were: undiluted serum, twofold-diluted serum, serum spiked with 120 mM NaCl and serum spiked with 300 mM NaCl (final concentrations). Microneedle sensor readings for K⁺ and Na⁺ were compared with values obtained using inductively coupled plasma mass spectrometry (ICP-MS; Agilent 8900), followed by grid error analysis. For pH sensing, the conditions included: undiluted serum, serum mixed 1:1 (v/v) with pH 4 buffer (BX16341, Sigma-Aldrich), serum mixed 1:1 with pH 5 buffer (BX16551, Sigma-Aldrich) and serum mixed 1:1 with pH 10 buffer (BX16361, Sigma-Aldrich). Results were compared to measurements from a commercial pH metre (edge Dedicated pH/ORP metre), followed by grid error analysis. For glucose sensing, rat kidney homogenate samples were tested under the following conditions: undiluted, twofold-diluted, fourfold-diluted and spiked with 36 mg dl⁻¹ glucose (final concentration). Results of sensor data were compared with those of a commercial glucose metre (Abbott FreeStyle Lite), and Clarke grid error analysis was performed. For lactic acid sensing, undiluted, twofold-diluted, fourfold-diluted rat kidney homogenate samples and a sample spiked with 5 mM lactic acid (final concentration) were used. Results were compared with a commercial test kit (EDGE Lactate Test kit), followed by grid error analysis. For uric acid sensing, the same dilution scheme was used, with an additional sample spiked with 0.3 mM uric acid (final concentration). Sensor outputs were compared with those from a commercial uric acid monitor (SpeedGUC 3-in-1 Multifunction Uric Acid Test gout monitor), and grid error analysis was performed.

Chronic in vitro test of the sensors

Long-term stability experiments for K⁺, glucose, oxygen and EMG sensors were conducted in a kidney homogenate environment at 37 °C. The voltage, current and impedance signals from each sensor were separately measured and analysed.

Microneedle insertion test in rat kidneys

A high-precision miniature S-Beam load cell (FUTEK, FSH03869), paired with a USB output module providing a resolution of 7 µN (FUTEK, FSH04720), was used to quantify the resistance force during microneedle insertion and retraction in fresh ex vivo rat kidneys. Bare and barbed microneedles were securely mounted on a stereotactic frame and inserted perpendicularly into the kidney tissue at a controlled rate of 5 mm min⁻¹, followed by retraction at the same speed.

Finite element analysis

The deformation process and tissue adhesion behaviour of the backward-facing barbs were numerically investigated using the solid mechanics module of COMSOL Multiphysics. For the deformation process, a two-dimensional symmetric model was created. A line segment was defined as the contact surface, with angles of 15°, 25° and 35° relevant to the y axis. The contact line underwent a fixed displacement

to simulate the deformation process. The deformation and stress distribution of the barbs after contact were computed. To characterize tissue adhesion of the barbs, the microneedle was treated as a rigid body and the tissue was set as a linear elastic material. The main boundary of the model was set such that the bottom side was fixed, a pair of equal tension forces was applied on both sides to simulate the tissue tension, and a gradually increasing force was applied on the upper part of the microneedle to simulate the extraction process. The PLGA interconnects were characterized by a density of $1,530 \text{ kg m}^{-3}$, a Young's modulus of 32.0 MPa and a Poisson's ratio of 0.12 (ref. 89).

Animal experiments

Experimental protocols were approved by the Institutional Animal Care and Use Committee (IACUC) of Dartmouth College. All animals were handled following the guidelines for the care and use of laboratory animals described by the IACUC of Dartmouth College (Protocol #00002330). Male Sprague–Dawley rats weighing $250\text{--}300 \text{ g}$ (purchased from Charles River Laboratories) were used for experiments. Rats were housed in a climate-controlled room with a 12-h light/12-h dark cycle and fed ad libitum.

Device implantation procedure

Sprague–Dawley rats were anaesthetized with 2% isoflurane gas delivered via a nose cone using an anaesthesia system (Isotec 4, SurgiVe). All animals were subjected to a 12-h fasting period with access to water before surgery. Animals were placed on a thermostatic heating pad to maintain normothermia throughout the procedure. Before surgery, abdominal hair was removed using a depilatory cream (VEET Hair Removal Cream, Reckitt Benckiser). A laparotomy was performed via either a ventral midline or flank incision to access the abdominal cavity. After identifying the target organ, the device was mounted in place. Sufficient slack in the e-suture was left within the abdominal cavity to prevent displacement of the microneedle device due to abdominal movement. The abdominal wall was partially closed using approximately three turns of the e-suture. The incision was then closed in layers with standard sutures (non-bioresorbable). Next, the skin was first partially closed using the remaining e-suture (approximately two turns), followed by complete closure with standard sutures (non-bioresorbable). Finally, the distal end of the e-suture was unrolled and connected to the external electronics. The use of standard sutures reduces scars that can be caused by bioresorbable sutures.

Bioresorption studies in rats

Devices were implanted according to the standard implantation procedure ($n = 4$ rats). At 1 week post implantation, the devices were triggered at 2.5 V to initiate dissolution of the gold coatings. One rat was euthanized each subsequent week to assess the morphology of the implanted device. In a separate cohort ($n = 4$ rats), the e-suture was implanted subcutaneously. The overlying skin was surgically reopened weekly to inspect the condition of the e-suture.

Acute kidney ischaemia monitoring

Devices were implanted using the standard implantation procedure ($n = 3$ rats), with suture steps omitted. Rats were euthanized at the conclusion of the experiments. Renal ischaemia was induced at 15-min intervals by occluding the renal artery and vein using haemostatic forceps. Electrochemical signals were recorded every 5 min. A 3×3 microneedle device was subsequently used to map spatiotemporal changes in renal oxygenation under the same ischaemia simulation protocol.

Chronic kidney ischaemia monitoring

Devices were implanted according to the standard implantation procedure ($n = 2$ rats). In the ischaemia group, chronic renal ischaemia was induced by suturing the blood vessels of the left kidney to occlude

blood flow. The implanted devices monitored glucose concentrations in the kidney over a 7-day period. To modulate renal glucose levels, intraperitoneal injections of 20 wt% glucose were administered on days 0, 3 and 7. Reference blood glucose measurements were obtained using a commercial glucometre (Abbott FreeStyle Lite) with tail-tip blood samples collected at 10-min intervals.

Gut monitoring

Rats were prepared following the implantation procedure. A segment of the small intestine was extruded, and a device was wrapped around the jejunum. The microneedle electrodes minimally invasively pierced into the lumen of the intestine. Electrochemical signals for glucose, Na^+ , K^+ and pH were recorded at 5 min/10 min intervals, and at the 20th min of the experiment, 2 ml 20 mM glucose/PBS solution was injected to study intestinal absorption. Subsequent recordings were made until the 90th min. After the completion of the experiment, the animals were euthanized. For myoelectricity experiments in the intestine, the microneedles were located within the intestinal wall without puncturing the intestine. Two sets of experiments were conducted: in the first set, 1 ml of PBS was injected first, followed by injection of 1 ml 20 mM glucose/PBS solution; in the other set, 1 ml of PBS was injected first, followed by injection of 1 ml capsaicin/PBS solution ($100 \text{ }\mu\text{M}$).

In vivo evaluation of biocompatibility

To assess inflammation at the microneedle insertion sites, devices with PLGA/Au and crude 3D-printed microneedles were implanted in the kidney ($n = 3$ rats per group: 3-day PLGA/Au, 7-day PLGA/Au and 3-day 3D-printed). Kidney tissues were collected on days 3 and 7, fixed, paraffin embedded, sectioned and stained with H&E. For haematologic and clinical chemistry assessments, blood samples were collected from sham and device-implanted rats at 2 and 4 weeks post implantation ($n = 3$ rats per group) via tail vein puncture. For complete blood counts, $500\text{--}600 \text{ }\mu\text{l}$ of blood was collected into K2-EDTA tubes. For serum chemistry, $500\text{--}600 \text{ }\mu\text{l}$ was collected into uncoated tubes. All analyses were performed by IDEXX BioAnalytics Diagnostic Laboratories. Major organs were collected at 4 weeks for trace element analysis (S, Mo and Au) using Agilent 8900 ICP-MS. In separate cohorts of sham and device groups ($n = 3$ rats per group), body weight was monitored weekly, and major organs (heart, lungs, liver, kidneys, spleen and intestines) were collected and weighed at 5 weeks. For long-term biocompatibility evaluation, additional groups ($n = 3$ rats per group: sham, 1-month device and 6-month device) were euthanized at 1 and 6 months, and major organs were collected and stained with H&E.

Mechanical stability test using a laboratory shaker

Devices were implanted according to the standard implantation procedure ($n = 3$ rats) and rats were euthanized upon completion of surgery. Euthanized rats were fixed on a laboratory shaker (VWR) and subjected to high-speed shaking for 1 h. The acceleration and angular velocity data were measured using an inertial measurement unit, BMI160 (Bosch Sensortec).

Microscale CT imaging

CT imaging was performed using a preclinical microPET/CT imaging system (Quantum GX3 microCT System) with 'high resolution' magnification, $68.0 \text{ }\mu\text{m}$ voxel size, 100 kV, $200 \text{ }\mu\text{A}$ and 671 mGy .

Reporting summary

Further information on research design is available in the Nature Portfolio Reporting Summary linked to this article.

Data availability

All data supporting the findings of this study are presented in the Article and its Supplementary Information. Source data are provided with this paper.

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Author contributions

X.L. and W.O. conceived the ideas and designed the research. X.L. developed the sensors. X.L., G.L., M.Z., J.R. and M.M. manufactured and tested the sensors. S.L. designed and manufactured the electronics. J.M. and C.Y. performed the finite element simulation. X.L. performed the sensor characterizations and animal experiments. W.O. and H.F. supervised the research. X.L. and W.O. wrote the manuscript. All authors reviewed and commented on the manuscript.

Competing interests

The authors declare no competing interests.

Additional information

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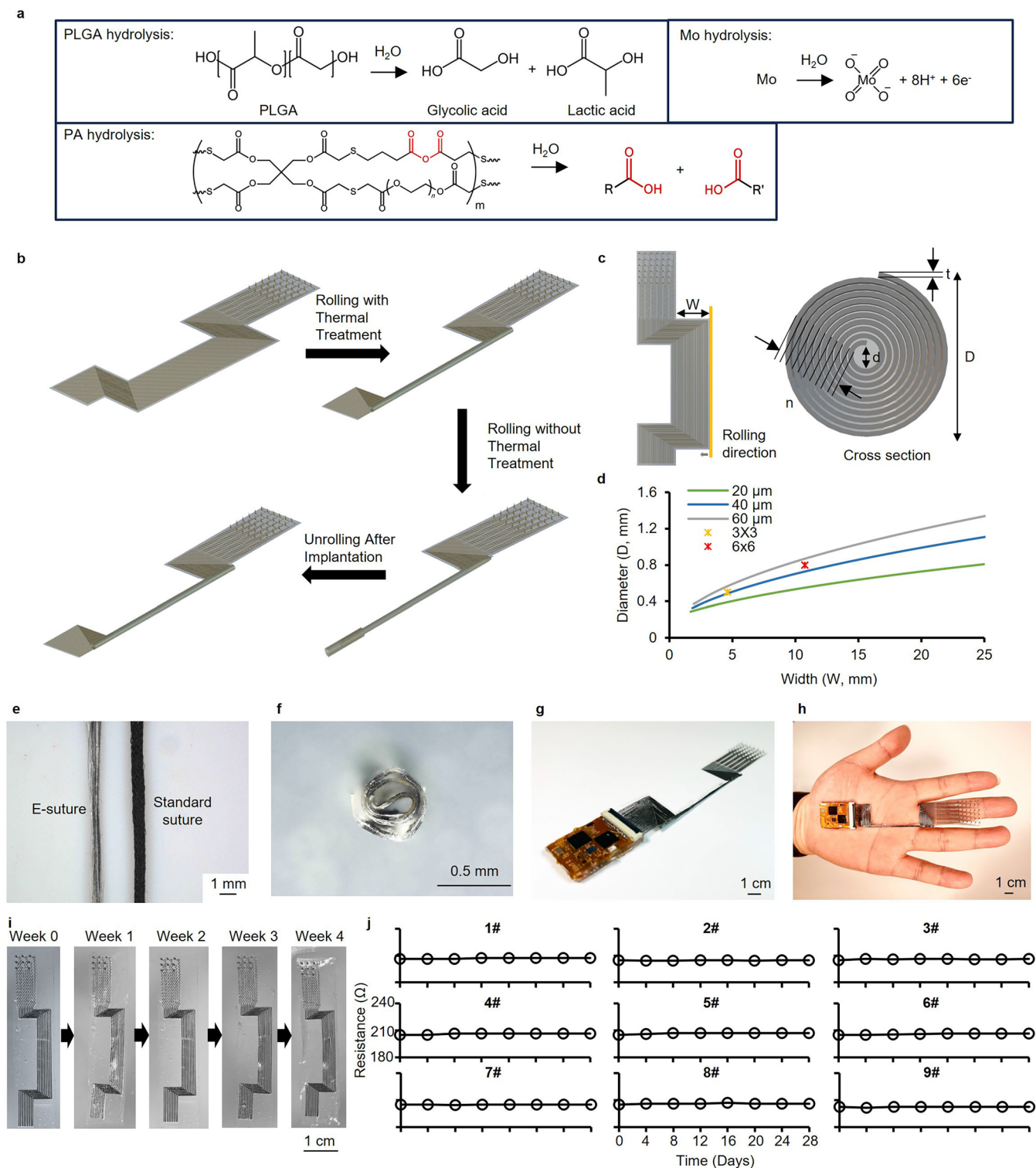
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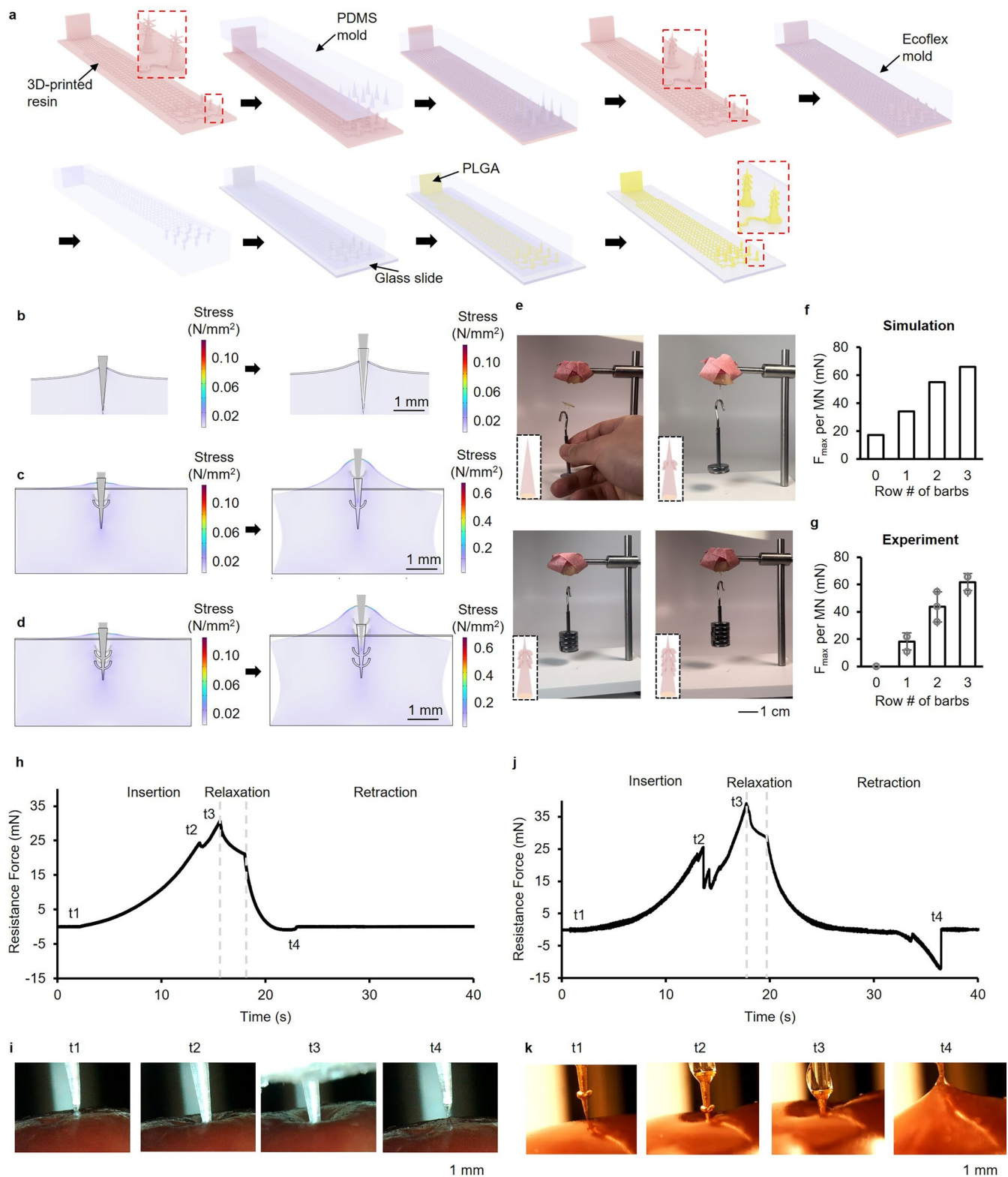
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Extended Data Fig. 1 | Additional data on design concepts and system features.

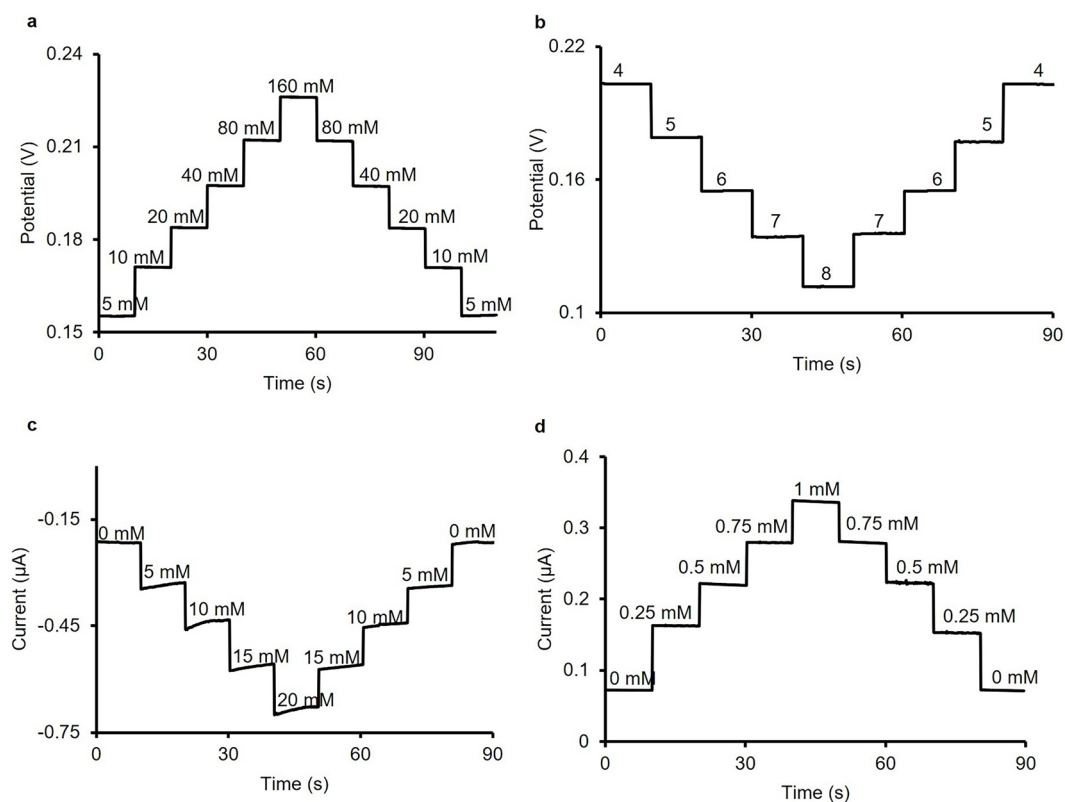
a, Bioresorption processes of materials. **b**, Schematic illustration of the rolling process of the e-suture. **c**, Definition of rolling parameters. **d**, E-suture diameter v.s. total width of electrical interconnects at different substrate thickness. **e**, Micrograph of the e-suture and a standard size #4 suture.

f, Cross-sectional micrograph of the e-suture. **g**, Photograph of a completed 6×6 device. **h**, Photograph of a completed 6×6 device on the palm. **i**, Stability of a 3×3 device in PBS at 37 °C. **j**, The electrical resistance of individual electrical interconnects of the e-suture (1-9#) in PBS at 37 °C.

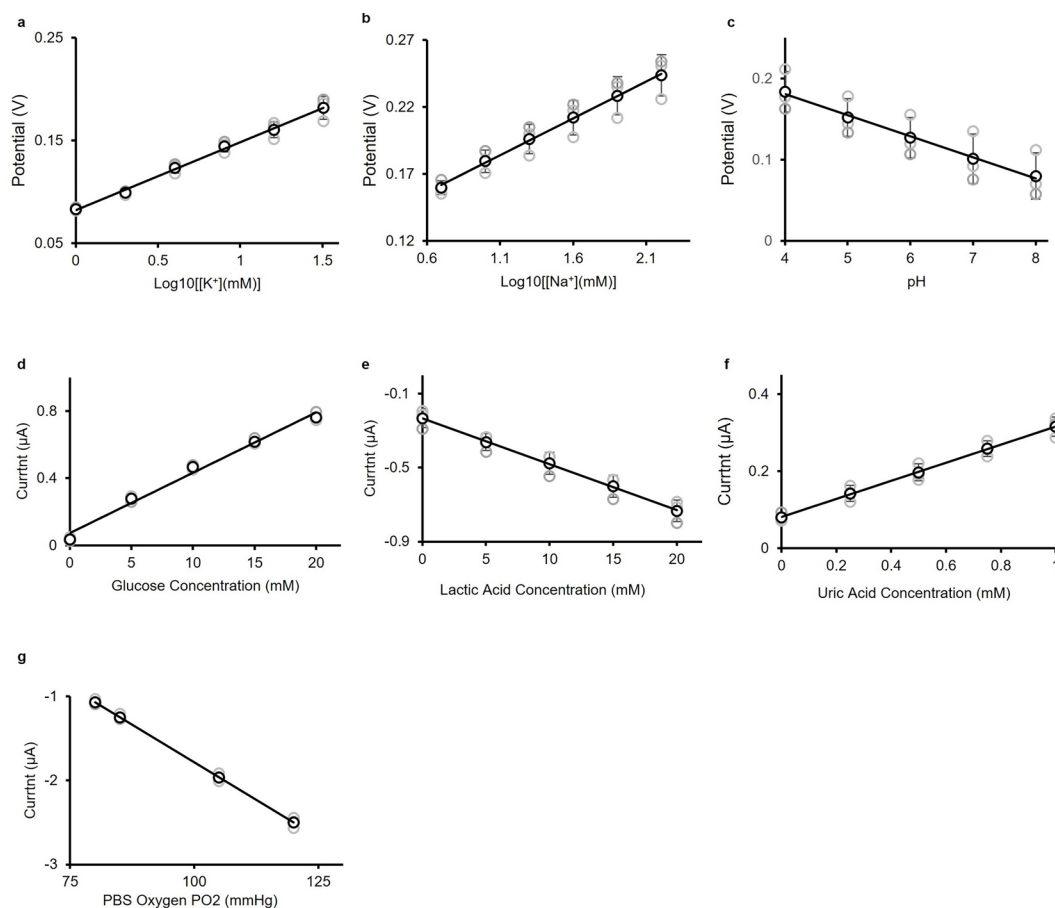


Extended Data Fig. 2 | Mechanical characterizations of the barbed microneedle array. **a**, Schematic illustration of the fabrication process of barbed microneedles. **b-d**, Numerical simulation of the tissue retention characteristics of microneedles with 0 (**b**), 1 (**c**), and 2 (**d**) rows of barbs. **e**, Photographs of weight-holding tests of microneedles with 0, 1, 2, and 3 rows of barbs using 10-gram weights. **f**, Simulated maximum pull-out force of barbed microneedles. **g**, Maximum pull-out force of barbed microneedles

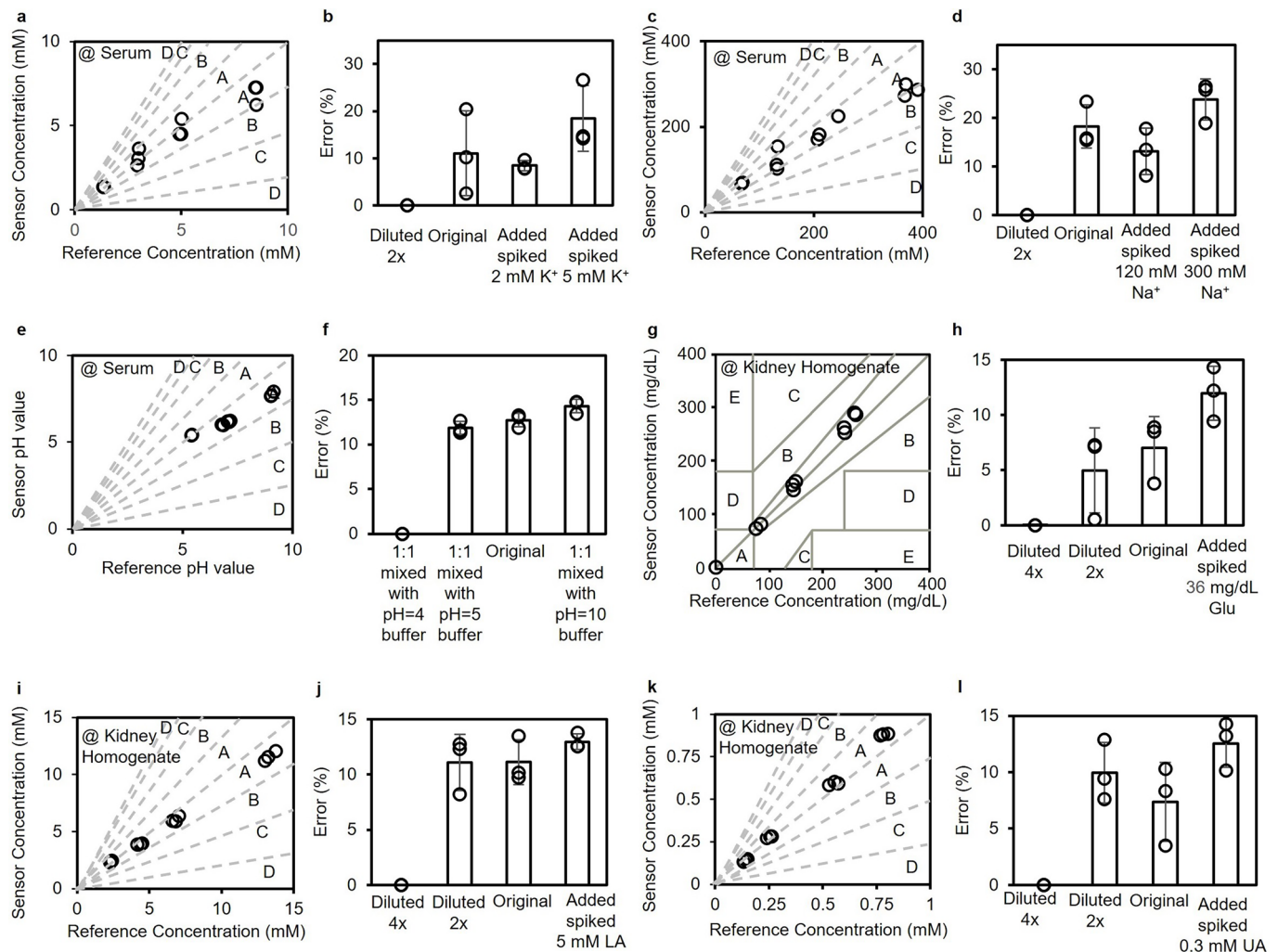
measured by the weight-holding test. The data are presented as mean $\pm$ s.d. ($n = 3$ independent experiments). **h**, Resistance force experienced by a bare microneedle during an insertion test in a rat kidney. **i**, Photographs of the insertion test of a bare microneedle. **j**, Resistance force experienced by a barbed microneedle during an insertion test in a rat kidney. **k**, Photographs of the insertion test of a barbed microneedle.



Extended Data Fig. 3 | Response and reversibility of microneedle electrochemical sensors. a, Na^+ sensor. b, pH sensor. c, Lactic acid sensor. d, Uric acid sensor.

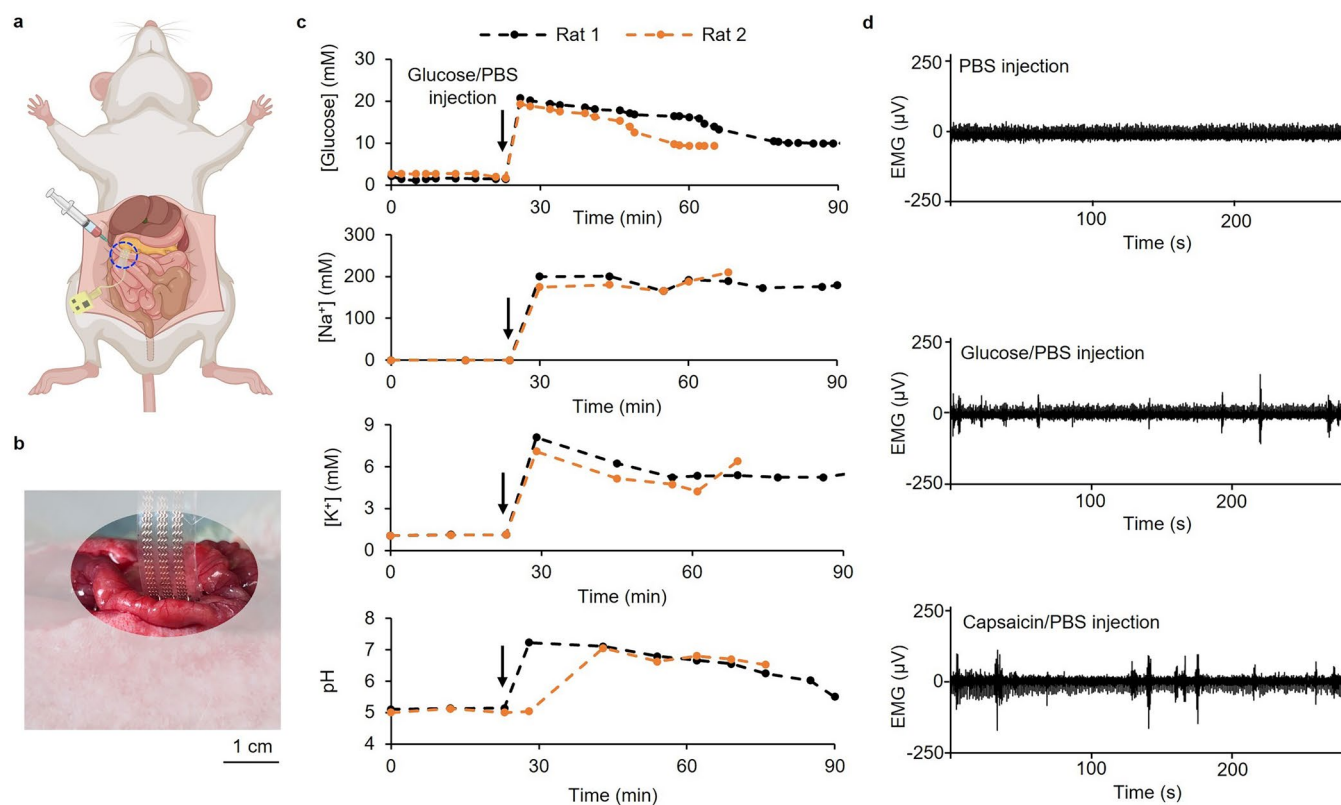


Extended Data Fig. 4 | Reproducibility of microneedle electrochemical sensors (n = 3 independent sensors). a, K^+ sensor. b, Na^+ sensor. c, pH sensor. d, Glucose sensor. e, Lactic acid sensor. f, Uric acid sensor. g, Oxygen sensor. All the data are presented as mean $\pm$ s.d.



Extended Data Fig. 5 | Validation of microneedle electrochemical sensors against standard methods. a–b, Error grid analysis (a) and relative errors to reference values (b) of the K^+ sensor. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors). **c–d,** Error grid analysis (c) and relative errors to reference values (d) of the Na^+ sensor. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors). **e–f,** Error grid analysis (e) and relative errors to reference values (f) of the pH sensor. Region A corresponds to those values within $<20\%$ deviation from the reference results, which could inform reliable decisions. Region B shows inaccurate values with 20% – 50% deviation from the reference results. Region C reflects inaccurate values with 50% – 80% deviation. Region D shows inaccurate values indicating a potential failure to detect target chemicals. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors). **g–h,** Clarke's error grid analysis (g) and relative errors to reference values (h) of

the glucose sensor. Region A corresponds to those values within 20% deviation from the reference glucose values. Region B shows inaccurate values with $>20\%$ deviation from the reference glucose values but would not lead to inappropriate diabetes treatment. Region C reflects inaccurate values leading to unnecessary diabetes treatment. Region D shows inaccurate values indicating a potential failure to detect hypoglycemia or hyperglycemia. Region E corresponds to those inaccurate values that would confuse treatment of hypoglycemia for hyperglycemia and *vice versa*. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors). **i–j,** Error grid analysis (i) and relative errors to reference values (j) of the lactic acid sensor. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors). **k–l,** Error grid analysis (k) and relative errors to reference values (l) of the uric acid sensor. The definitions of the regions are the same as those in a–f. The data are presented as mean $\pm$ s.d. ($n = 3$ independent sensors).



Extended Data Fig. 6 | Monitoring of gut disorders in rats. a, Schematic illustration of gut monitoring using the device. Created in BioRender. Ouyang, W. (2025) <https://BioRender.com/21omdf7>. **b**, Photograph of the SMART

encircling the small intestine of a rat. **c**, Concurrent monitoring of glucose, Na^+ , K^+ , and pH in the lumen of the intestine. **d**, EMGs of the intestine upon injection of PBS, glucose, and capsaicin.

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☒	☐ Plants

Methods

n/a	Involved in the study
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☒	☐ Flow cytometry
☒	☐ MRI-based neuroimaging

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Laboratory animals	All experiments used adult Sprague-Dawley rats (250-350 g, 8-12 weeks old at start of experiments) from Charles River Laboratories (Wilmington, MA).
Wild animals	No wild animals were used in the study.
Reporting on sex	The findings from our device development and validation effort apply equally to both sexes. Fifty-six Sprague-Dawley rats were used for these studies. No sex-based analyses were performed, as the device is agnostic to signal inputs and both sexes have comparable physiological responses.
Field-collected samples	No field collected samples were used in the study.
Ethics oversight	All animals were handled following the guidelines for the care and use of laboratory animals described by the Institutional Animal Care and Use Committee of Dartmouth College (Protocol # 00002330).

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Plants

Seed stocks	No plants were used in this study.
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Authentication	No plants were used in this study.