Development/Plasticity/Repair

Depolarizing GABAergic Conductances Regulate the Balance of Excitation to Inhibition in the Developing Retinotectal Circuit In Vivo

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Neurotransmission during development regulates synaptic maturation in neural circuits, but the contribution of different neurotransmitter systems is unclear. We investigated the role of GABA_A receptor-mediated Cl^- conductances in the development of synaptic responses in the Xenopus visual system. Intracellular Cl^- concentration ([Cl^-]i) was found to be high in immature tectal neurons and then falls over a period of several weeks. GABAergic synapses are present at early stages of tectal development and, when activated by optic nerve stimulation or visual stimuli, induce sustained depolarizing Cl^- conductances that facilitate retinotectal transmission by NMDA receptors. To test whether depolarizing GABAergic inputs cooperate with NMDA receptors during activity-dependent maturation of glutamatergic synapses, we prematurely reduced [Cl^-]i in tectal neurons in vivo by expressing the Cl^- transporter KCC2. This blocked the normal developmental increase in AMPA receptor-mediated retinotectal transmission and increased GABAergic synaptic input to tectal neurons. Therefore, depolarizing GABAergic transmission plays a pivotal role in the maturation of excitatory transmission and controls the balance of excitation and inhibition in the developing retinotectal circuit.

Key words: synaptic development; activity-dependent; intracellular chloride; GABAergic transmission; glutamatergic transmission; visual system

Introduction

The strength, distribution, and type of synaptic inputs determine the behavior of a neuron within a neural circuit. For instance, experiments in sensory systems illustrate that the balance of excitatory and inhibitory connections dictates how a neuron integrates and processes information (Wehr and Zador, 2003; Gabriel et al., 2005). The mechanisms controlling the development of excitatory and inhibitory inputs are still unclear, but it is widely believed that neural activity is important. Activation of NMDA receptors (NMDARs) plays a central role in the development of excitatory synapses and circuits, including the retinotectal system (Rajan et al., 1999; Aamodt et al., 2000). During glutamatergic synaptic development, NMDARs are thought to be present and functional at nascent synapses, whereas AMPA receptors (AMPARs) are trafficked to, and stabilized at, synaptic sites as the cell matures (Wu et al., 1996; Isaac et al., 1997; Xiao et al., 2004). Trafficking AMPARs into NMDAR-only synapses requires NMDAR activity, Ca^{2+} influx, and calcium/calmodulin-dependent kinase II activity (Pettit et al., 1994; Shi et al., 1999; Zhu and Malinow, 2002). Because of the voltage-dependent Mg^{2+} block of the NMDAR, one open question concerns the source of membrane depolarization required for NMDAR activity under conditions when AMPAR activity is low.

During development, GABA_A receptor (GABA_AR) activation by GABA, the traditional inhibitory transmitter, can provide significant membrane depolarization (Ben-Ari, 2002; Owens and Kriegstein, 2002). This depolarization results from the immature expression of Cl^- transporters that generate a relatively positive Cl^- reversal potential (E_{Cl^-}), and its consequences depend on the degree of depolarization and the nature of other active conductances. For instance, in immature hippocampus, exogenous application of GABA_A agonists can decrease the Mg^{2+} block of single NMDARs and increase NMDAR-mediated Ca^{2+} influx (Leinekugel et al., 1997). Therefore, during network activity in the developing brain, postsynaptic depolarizing GABAergic Cl^- conductances may provide a source of membrane depolarization that relieves the NMDAR Mg^{2+} block and may be required for NMDAR-mediated developmental increases in AMPAR-mediated transmission.

In vivo pharmacological and genetic manipulation experiments indicate that GABAergic activity is required for synaptic circuit development (Hensch et al., 1998; Tao and Poo, 2005). Here we investigate the impact of postsynaptic GABAergic conductances on synaptic development in the retinotectal system of Xenopus laevis. We show that E_{Cl^-} is depolarizing around the

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time of retinal afferent innervation and that retinal activity evokes robust GABA, R-mediated Cl\(^-\) conductances that are ideally suited to facilitate NMDAR transmission. Recent experiments in dissociated neurons reveal that early expression of the Cl\(^-\) transporter KCC2 can shift \(E_{Cl} \) (Lee et al., 2005) and augments GABAergic synaptic development (Chudotvorova et al., 2005). To test whether depolarizing GABAergic Cl\(^-\) conductances are required for normal synaptic development in the retinotectal circuit, we manipulated \(E_{Cl} \) in vivo by premature expression of KCC2. Tectal neurons that had a mature \([Cl^-]\), imposed failed to develop normal glutamatergic inputs but acquired a stronger contribution from GABAergic inputs. These experiments support a model in which early GABAergic inputs cooperate with NMDARs to mediate developmental plasticity of glutamatergic synapses and the balance of excitatory and inhibitory circuits within the retinotectal system.

**Materials and Methods**

*Electrophysiology.* Albino *X. laevis* tadpoles were reared under controlled conditions (20°C, 12 h light/dark cycle), and embryo stages were consistent with published criteria (Nieukoop and Faber, 1994) (4 d, stage 40; 8 d, stage 47; 16 d, stage 48/49; 52 d, stage 50/51). For whole-brain recordings, tadpole brains were prepared as described previously (Wu et al., 1996). Animals were anesthetized in 0.02% tricaine methanesulfonate (MS-222), and brains were cut along the dorsal midline to expose the ventricular surface and dissected into extracellular saline (115 mM NaCl, 2 mM KCl, 3 mM CaCl\(_2\), 1.5 mM MgCl\(_2\), 5 mM HEPES, 10 mM glucose, and 10 mM glycerin, pH 7.2, osmolality of 255 mosm). Tissues were soaked in saline solution for at least 15 min before recording experiments began. The solutions used for recording were prepared from a stock solution (5 mg/ml) that was consistent with the intracellular NaCl concentration. Brains were either removed from the skull (Zhang et al., 1998; Aizenman et al., 2003). Visual stimuli consisted of monocular focal field flashes from a computer-controlled green light-emitting diode (Amex 567 nm; Allied Electronics, Fort Worth, TX) presented to the contralateral eye. Responses were recorded to 1 s OFF stimuli, presented at a frequency of 0.05 Hz.

To compare across different ages, we recorded from neurons that were rostral to the proliferative zone, in the central 50% of the tectum. Recordings were made using glass micropipettes (5–9 M\(\Omega\)) filled with the appropriate intracellular solution (pH 7.2, osmolality of 255 mosm). All reported voltages were corrected for the liquid junction potential (LJP) that was measured between the intracellular and extracellular solution. A stabilized microelectrode holder was used to minimize pipette drift (G23 Instruments, London, UK). Neurons had input resistance in the range 1–4 G\(\Omega\) and series resistance \(<50 M\Ω\), which were monitored throughout the experiments. Signals were measured with an Axopatch 200B, Axoclamp 2B, or Multiclamp 700A amplifier (all from Molecular Devices, Palo Alto, CA) and digitized using a Digidata 1200 or 1322A analog-to-digital board (Molecular Devices). Stimulation and data acquisition was performed with pClamp8 software (Molecular Devices) and digitized at 10 kHz. Data was analyzed off-line using custom Matlab software (MathWorks, Natick, MA) and pClamp9 (Molecular Devices). Unless otherwise stated, drugs and other reagents were obtained from Sigma (St. Louis, MO). As required, the following antagonists and blockers were added to the extracellular solution: CNQX (20 \(\mu M\); Tocris Cookson, Ballwin, MO), SR95531 [2-(3-carboxypropyl)-3-amino-6-(4-methoxyphenyl)pyridazinone bromide] (5 \(\mu M\); Tocris Cookson), APV (100 \(\mu M\); Tocris Cookson), strychnine (60 \(\mu M\); tetradotoxin (TTX) (1 \(\mu M\); Alomone Labs, Jerusalem, Israel), and CGP35584 [(-)-6-carboxy-methyl-phenylacetic acid] (50 \(\mu M\); Tocris Cookson), QX-314 [2-(3,5-dimethylphenyl)etacetic acid] (2 \(\mu M\); Alomone Labs) was added to the intracellular recording solution. Data were tested for normality using a Kolmogorov–Smirnov test. Those deemed normal were compared using parametric tests (t test, ANOVA), and those deemed non-normal were compared using nonparametric tests (Mann–Whitney \(U\) test, Kruskal–Wallis test). All tests were two-tailed.

**Gramicidin recordings.** For gramicidin recordings, we used a K\(^+\)–based intracellular solution with either low \([Cl^-]\); solution 1 (in mM): 114 K-glucuronate, 1 MgCl\(_2\), 20 HEPES, 10 EGTA, 2 ATP, and 0.3 GTP (LJP of 12 mV) or high \([Cl^-]\); solution 2 (in mM): 114 KCl, 1 MgCl\(_2\), 20 HEPES, 10 EGTA, 2 ATP, and 0.3 GTP (LJP of 4 mV). These intracellular solutions generate a substantial shift in \(E_{Cl} \), on moving from perforated to whole-cell mode. Gramicidin (Calbiochem) was dissolved in dimethylsulfoxide to produce a stock solution (5 mg/ml). Every 3 h, 4 \(\mu\)l of this stock was added to 1 ml of prefiltered intracellular solution and sonicated for 30 s to produce a final gramicidin concentration of 20 \(\mu\)g/ml. TTX was added to the extracellular solution to block voltage-gated sodium currents and action-potential activity. After formation of a tight membrane seal, perforation was evaluated by monitoring the decrease in access resistance, which usually stabilized between 8 and 20 min (Kyrozis and Reichling, 1995). After measurement of \(E_{Cl} \), the integrity of the perforated patch was confirmed by applying suction to rupture the underlying plasma membrane and observing both a decrease in access resistance and a shift in \(E_{Cl} \) that was consistent with the intracellular solution. In both perforated and whole-cell recordings, access resistance was determined by analyzing transient responses to voltage-clamp steps, and all membrane potential (\(V_{m} \)) measurements were subsequently corrected for the voltage drop across the series resistance. \(E_{Cl} \) was calculated from the amplitude of muscimol-evoked (50 \(\mu M\); Tocris Cookson) currents at different command potentials. We assumed that, unlike with nystatin and amphotericin, gramicidin patches lacked a permeant anion on either side of the sealed membrane and therefore a Donnan potential was unlikely to develop (Kyrozis and Reichling, 1995). Repeated agonist application can influence the apparent \(E_{Cl} \), particularly in immature neurons in which Cl\(^-\) regulation mechanisms are less effective (Ehrlich et al., 1999). Therefore, the interval between agonist applications was made sufficiently long (30–60 s) to avoid shifts in \(E_{Cl} \), even in the youngest neurons (data not shown). Agonists were applied through a patch pipette connected to a picospirator (5–10 psi for 20–50 ms; General Valve, Fairfax, NJ), adjusted such that current amplitudes did not exceed 200 pA. Peak currents were plotted as a function of voltage, and \(E_{Cl} \) was defined as the x-intercept value of the current–voltage curve. Recordings were performed in HEPES-buffered saline, and \(E_{GABA} \) was confirmed as an appropriate measure of \(E_{Cl} \) by establishing that the \(E_{GABA} \) measured in whole-cell configuration with different intracellular Cl\(^-\) solutions was predicted by the Nernst equation. Separate experiments in a bicarbonate-buffered intracellular solution revealed that \(E_{GABA} \) in the tectal tectum, supporting the conclusion that high \([Cl^-]\) underlies and dominates the depolarizing GABA, R responses in immature tectal neurons (data not shown). Resting membrane potential (RMP) was measured in whole-cell current-clamp recordings with intracellular solution 2, minus gramicidin.

**Isolating conductances.** Synchronized evoked glutamate receptor currents and Cl\(^-\) currents were recorded using a Cs\(^+\)–based intracellular solution [solution 3 (in mM): 114 Cs-methane sulfonate, 1.5 MgCl\(_2\), 3 tetraethylammonium-Cl, 20 HEPES, 10 EGTA, 2 ATP, and 0.3 GTP (LJP of 10 mV)] and were isolated by voltage clamping at the reversal potential of the currents. Cl\(^-\) conductances were isolated and measured as outward currents at a holding potential (\(V_{h} \)) of 0 mV (the reversal potential for glutamatergic currents under these conditions). Meanwhile, AMPA conductances were isolated and measured as inward currents at a \(V_{h} \) of −70 mV (just above the reversal potential for CI\(^-\) currents and at which NMDARs exhibit strong open channel block by external Mg\(^{2+}\)). To reveal AMPA–GABA\(_{A} \) sequences within the same trial (see Fig. 2C), we added APV extracellularly and recorded at a \(V_{h} \) intermediate to the glutamatergic and Cl\(^-\) current reversal potentials (−40 mV). When AMPA, GABA\(_{A} \), and NMDA conductances were isolated in the same cell (see Fig. 4A), we recorded AMPA and GABA\(_{A} \) conductances (\(V_{h} \) of −70 and 0 mV, respectively), and then applied SR95531 extracellularly and recorded the combined AMPA and (unblocked) NMDA conductance as an tested for normality using a Kolmogorov–Smirnov test. Those deemed normal were compared using parametric tests (t test, ANOVA), and those deemed non-normal were compared using nonparametric tests (Mann–Whitney \(U\) test, Kruskal–Wallis test). All tests were two-tailed.
outward current at a \( V_h \) of -45 mV. In experiments in which nMDA responses were isolated from all other conductances and recorded at voltages between -80 and +50 mV (see Figs. 4 B, 5D), CNQX, SR95531, and strychnine were added to the extracellular solution and QX-314 was added to the intracellular solution. CGP55845 was added extracellularly in some experiments but had no effect on the evoked currents. For all experiments using bursts of afferent stimulation (one to three stimuli, 50 Hz), for each cell, the stimulation intensity was set such that a single stimulus evoked a monosynaptic AMPA current of 50–100 pA at a \( V_h \) of -70 mV. For minimal stimulation experiments, the stimulus intensity was gradually increased from zero to the point at which stable, nonfailures of transmission were observed. These synaptic responses are believed to represent the transmission from a single retinal axon, although each axon can make multiple synaptic contacts with a single tectal cell (Wu et al., 1996). Current onset latencies were defined as the time from the stimulus artifact to 5% of the peak current. Conductances were calculated by dividing the current by the driving force.

**Dialysis and dynamic-clamp experiments.** To impose an "immature" or "mature" \([\text{Cl}^-]\), by dialysis, two different intracellular solutions were used: the first with 26 mM \([\text{Cl}^-]\) (solution 4 in mM): 90 K-glucuronate, 1.5 MgCl2, 20 HEPES, 10 EGTA, and 0.3 GTP (LJP of 10 mV) and the second with 11 mM Cl\(_-\) (solution 5 in mM): 104 K-glucuronate, 3 KCl, 1.5 MgCl2, 20 HEPES, 10 EGTA, and 2 ATP, and 0.3 GTP (LJP of 11 mV). For dynamic-clamp recordings, we blocked GABA\(_R\)s only in the recorded cell by using a \( \text{K}^+ \)-based intracellular solution containing 4,4′-disothiocyanostilbene-2,2′-disulfonic acid (DIDS) and \( I_{\text{Ca}}^+ \) (solution 5 in mM): 105 potassium fluoride, 8 KCl, 20 HEPES, 10 EGTA, and 0.5–1 DIDS (LJP of 7 mV) (Nelson et al., 1994). DIDS was made as a 500 mM stock in DMSO, and 1–2 mM was added to 1 ml of intracellular solution. DIDS was used at the earliest developmental stages and therefore was unlikely to affect any endogenous KCC2 activity. Control experiments established the ability of the DIDS to block GABA\(_R\)s by confirming that it attenuated synchronously evoked Cl\(_-\) currents (94% block; 12 cells) (see Fig. 2A). Recordings of muscimol-evoked currents at different holding potentials supported the observation that the DIDS solution reduced the GABA\(_R\) conductance (90% block; nine cells; data not shown). In some cells, dynamic clamp was performed with intracellular solution 4, and GABA\(_R\)s were blocked extracellularly with SR95531. For dialysis and dynamic-clamp experiments, neurons had input resistances in the range 1–4 GΩ and were subjected to the stimulus artifact to 5% of the peak current. Conductances were calculated as described previously (Wu et al., 1996) by comparing failure rates at \(-70\) mV + 45 mV under minimal stimulus conditions (30–100 trials at each \( V_h \)). Evoked AMPA amplitudes were derived from the nonfailure events at \(-70\) mV. For all miniature postsynaptic current (mPSC) recordings, TTX and APV were added extracellularly. To improve event detection and avoid interference between conductances, AMPA and GABA\(_A\) mPSCs were pharmacologically isolated and thus recorded from different cells. AMPA mPSCs were recorded as inward currents at \(-70\) mV in SR95531, and GABA\(_A\) mPSCs were recorded as outward currents at 0 mV in CNQX. Three to 10 min of recordings were analyzed from each cell, and mPSCs (peak amplitude, >3 pA) were detected with a template-matching algorithm. Total AMPA or GABA\(_A\) input (in pico-siemens) was calculated as the area under the average mPSC (in pico-siemens per second) multiplied by the mini frequency (in hertz) and is a measure of the mean input to the cell at any one moment. On entering whole-cell mode, we allowed 10 min for dialysis of the intracellular solution before collecting data. Under these whole-cell recording conditions, the reversal potential for glutamatergic and Cl\(_-\)-mediated currents, and therefore their driving force, were equivalent across the different conditions: the reversal potential for AMPAR-mediated glutamatergic currents was comparable in nontransfected cells (1.2 ± 0.6 mV; 25 cells) and in cells transfected with green fluorescent protein (GFP) (0.8 ± 1.0 mV; 20 cells), Y1087D (0.8 ± 1.2 mV; 8 cells), or KCC2 (0.9 ± 1.0 mV; 22 cells). Equally, the reversal potential for GABA\(_A\)-mediated currents under whole-cell recording conditions was comparable in nontransfected cells (–75.8 ± 5.8 mV; 16 cells) and in cells transfected with GFP (–77.4 ± 3.5 mV; 12 cells), Y1087D (–78.6 ± 3.3 mV; 13 cells), or KCC2 (–77.1 ± 2.9 mV; 18 cells). Off-line analyses also confirmed that the frequency and amplitude of synaptic currents was stable over the period of data collection.

**DNA constructs and transfections.** The cDNA clone for rat KCC2 was a generous gift from Kai Kaila (University of Helsinki, Helsinki, Finland). The *Xenopus* expression plasmid was a bidirectional PCS2 vector (containing two cytochrome oxidase promoters, one on each strand) and was a generous gift from David Turner (University of Michigan, Ann Arbor, MI). One multiple cloning site contained enhanced GFP (Clontech, Palo Alto, CA) and the other multiple cloning site contained (1) the open reading frame of KCC2 (“KCC2”), (2) the Y1087D mutation of KCC2 (“Y1087D”), or (3) was empty and served as a control (“GFP”). Y1087D was generated by replacing the tyrosine residue at position 1087 of KCC2 with an aspartate residue. This mutation inhibits basal transport activity of KCC2 in *Xenopus* oocytes by 80% and completely inhibits hypotonic-induced KCC2 activity (Strange et al., 2000). The mutation was made with the QuickChange XL Site-Directed Mutagenesis kit (Stratagene, La Jolla, CA) and confirmed with DNA sequencing. Tadpoles at 3.5–4.5 dpf were anesthetized (0.02% MS-222), and tectal neurons were transfected by microinjecting a DNA solution into the brain ventricle (100 nl; 2 µg/µl) and then targeting brief voltage pulses across the tectum (five exponential pulses, 1 s interval, time constant of 70 ms, field strength of 200–400 V/cm). Animals recovered from anesthesia within 10 min, and, 36 h later, GFP was detected and expression of KCC2 and Y1087D was confirmed by immunohistochemistry. Under these conditions, only a small minority of neurons distributed across the ventricular surface of the tectum (0.5–3%) were transfected, and no transfected neurons were observed in the retina. For immunohistochemistry, tadpoles were fixed in 4% paraformaldehyde, and 30 µm horizontal sections were cut. Sections were incubated with a rabbit anti-rat KCC2 polyclonal antibody (1:200), which was a generous gift from John Payne (University of California, Davis, CA; Upstate, Charleston, SC) and then a goat anti-rabbit Alexa Fluor-594 secondary antibody (1:1000; Invitrogen, Carlsbad, CA) and were mounted in 4′,6-diamidino-2-phenylindole Vectorshield (Vector Laboratories, Burlingame, CA).
notectal circuit development. First, to determine whether these criteria are met. Recordings were done in the whole-brain preparation, which preserves the neural circuitry while allowing direct access for both stimulation and pharmacological manipulation of the retinotectal system. We tested whether GABAergic synaptic connections are recruited by retinal axon stimulation. Synaptic currents were evoked in 4 dpf tectal cells by delivering single or short bursts of stimuli to the optic nerve. By making whole-cell voltage-clamp recordings with a relatively low intracellular Cl\(^-\) solution, we were able to separate glutamatergic currents from currents carried by Cl\(^-\) (see Materials and Methods). Recordings at the reversal potential for AMPA currents allowed us to monitor Cl\(^-\) currents, whereas recordings at E\(_{Cl^-}\) allowed us to monitor AMPA currents. Under these conditions, almost all tectal cells at 4 dpf exhibited monosynaptic AMPA currents resulting from activation of RGC afferents also exhibited sympathetically evoked outward currents at the AMPA reversal that were consistent with being carried by Cl\(^-\). Pharmacological experiments (Fig. 2A) in which glycine receptor and GABA\(_A\)R blockers were added to the bath demonstrated that, whereas the addition of 30 μM strychnine had little effect on these currents (8.1 ± 2.1% reduction), they were entirely blocked by the addition of the GABA\(_A\)R blocker SR95531 (97.1 ± 1.8% reduction; n = 6 cells). Consistent with this data, the Cl\(^-\) currents were selectively blocked by intracellular dialysis with a recording solution containing DIDS (94 ± 1.1% reduction compared with control solution; n = 12 and 9 cells, respectively) (Fig. 2A), a method that has been used previously to selectively block GABA\(_A\) conductances only in the recorded cell (Nelson et al., 1994). These experiments demonstrated that GABAergic synaptic connections are already present at 4 dpf and that they provide input onto the vast majority of tectal cells.

To further characterize the nature of these immature GABAergic synapses, we applied single stimuli to the optic chi-
Figure 2. Retinal activity drives a feedforward GABAergic circuit. A. Retinotectal synaptic responses were evoked by stimulation of RGC axons in the optic chiasm. SR95531, a GABA_A receptor antagonist, blocked the synaptically evoked Cl^- currents (n = 6 cells), but strychnine, a glycine receptor antagonist, did not. The Cl^- conductance was also blocked by intracellular dialysis of a recording solution containing DIDS (n = 12 and 10 cells for control and DIDS solutions, respectively). The stimulus artifact in the example traces and, in the following examples, has been truncated for clarity. B, GABA_A synaptic currents evoked by retinal afferent stimulation are delayed compared with AMPA currents. A single stimulus to the optic chiasm evoked a monosynaptic AMPA response (V_h of -70 mV) that was followed by a delayed GABA_A response (V_h of 0 mV) in the same tectal neuron (n = 15 cells). C, Minimal stimulation of the optic chiasm while recording tectal cell responses at an intermediate command potential (V_h of -40 mV) evoked AMPA–GABA_A sequences as well as trials in which the AMPA, the GABA_A, or both components of the response failed (n = 8 cells). D, These data are consistent with the existence of a disynaptic feedforward GABAergic circuit at these early stages of retinotectal development (4 dpf). Retinal afferents (RGC) provide direct monosynaptic glutamatergic input to tectal cells (TC), and GABAergic interneurons (IN) within this population of cells provide a source of feedforward GABAergic input.

GABAergic synaptic responses are efficiently recruited with multiple “burst-like” stimuli delivered to the optic nerve, and the resulting GABA_A-mediated conductances are both large and sustained. Pairs of stimuli delivered with varying interstimulus intervals (ISIs) showed that GABAergic input was enhanced if the optic nerve was activated sequentially within several hundred milliseconds and that this response was maximal for closely spaced stimuli such as might occur during a burst of retinal activity (Fig. 3A) (Tao et al., 2001). At an ISI of 20 ms, the response to a second stimulus was almost three times larger (2.9 ± 0.9-fold) than the response to one stimulus, but, at an ISI of 500 ms, there was no difference. Responses to trains of stimuli (one to three stimuli, 50 Hz) were used to directly compare the magnitude of GABA_A and AMPA conductances onto the same 4 dpf tectal neurons (Fig. 3B). Peak GABA_A conductance (mean of 1.1, 1.8, and 2.3 nS for one, two, and three stimuli, respectively) was slightly larger than the peak AMPA conductance (mean of 0.9, 1.3, and 1.6 nS, respectively), although not statistically significant (p = 0.68, 0.51, and 0.39 for one, two, and three stimuli, respectively; n = 9 cells). However, because of the sustained nature of the evoked GABA_A conductance, a dramatic difference was apparent in the integral of the two conductances. The GABA_A-integrated conductance (mean of 177.8, 524.4, and 1277.5 pS.s for one, two, and three stimuli, respectively) was substantially larger than the AMPA (mean of 4.6, 8.6, and 15.8 pS.s, p < 0.001, 0.001, and 0.001 for one, two, and three stimuli, respectively; n = 9 cells). Interestingly, the ratio of GABA_A/AMPA integrated conductance was larger for burst stimuli than for single stimuli (GABA_A/AMPA of 38.7, 60.9, and 80.9 for one, two, and three stimuli, respectively), indicating that, under strong retinal activity, tectal cells become increasingly dominated by the GABA_A conductance.

To test whether GABAergic inputs are activated by physiologically relevant stimuli and to compare responses between different stimuli, we performed in vivo recordings in which we presented visual stimuli to the intact retina of 4 dpf tadpoles (Fig. 3C). Whole-field dimming stimuli (OFF stimuli) were presented to the contralateral retina, and AMPA and GABA_A currents were isolated in tectal cells, as described above. Visual stimulation evoked robust AMPA and GABA_A responses that were comparable with those evoked by bursts of stimuli delivered to the optic nerve. The peak of the visually evoked GABA_A conductance...
evoked conductances were comparable with those evoked by GABAA conductances in tectal cells comparable with those evoked by short bursts of electrical stimuli to the RGC afferents (ISI of 20, 200, 400, and 800 ms; \(t\) factors substantially larger than the integrated AMPA conductance). This and all subsequent figures. J. Neurosci., May 10, 2006 • 26(19):5117–5130 Akerman and Cline

**GABAergic conductances in developing tectal neurons modulate voltage-dependent NMDAR block**

Addition of SR95531 during the experiments described above (Fig. 3B) enabled us to record a combined AMPA and NMDA conductance as an outward current at a positive \(V_m\) (+45 mV). Subtracting the AMPA conductance (recorded at ~70 mV) from the combined AMPA and NMDA conductance allowed us to isolate the three major synaptic conductances in the retinotectal circuit (AMPA, NMDA, and GABAA) within an individual neuron. A comparison of the three conductances revealed that the unblocked NMDA conductance at this age exhibits a much longer duration than the AMPA conductance and more closely parallels the temporal profile of the GABAA conductance (Fig. 4A). Consequently, the GABAA conductance at this stage of development has the appropriate temporal properties to modulate the \(V_m\) during the period that glutamate is bound to the NMDAR. NMDA responses in retinotectal synapses at 4 dpf exhibit the same voltage-dependent open channel block by external Mg\(^{2+}\) as has been observed in a variety of other systems (Fig. 4B). Peak inward current through the NMDAR was observed between ~40 and ~20 mV, which reflects the membrane potentials at which maximal net influx of permeable cations (Na\(^{+}\), K\(^{+}\), and Ca\(^{2+}\)) occurs and is close to the membrane potential at which peak Ca\(^{2+}\) influx occurs (Kovalchuk et al., 2000). The current–voltage relationship of the NMDAR in tectal cells was fit with an exponential (Fig. 4B) (see Materials and Methods), and this function was used to estimate the net inward current through the NMDAR from voltage waveforms recorded during periods of retinotectal circuit activity.

To test how developmental changes in \([Cl^-]\), impact NMDAR transmission, we adopted two different approaches that were designed to preserve all other conductances in the network while selectively manipulating \(Cl^-\) conductances only in the recorded cell. First, we dialyzed different intracellular \(Cl^-\) solutions into tectal cells and recorded their voltage responses during retinotectal circuit activity (Fig. 4C,D). Second, we used the dynamic-clamp recording technique (Robinson and Kawai, 1993; Sharp et al., 1993) to simulate realistic GABAA conductances with different \(E_{Cl^-}\), thereby simulating the developmental shift in \([Cl^-]\) within the same tectal cell (Fig. 5). The findings from both sets of experiments indicate that early \(Cl^-\) conductances are a key regulator of NMDAR transmission in the retinotectal system.

Whole-cell current-clamp recordings were made from 4 dpf tectal neurons with different internal solutions, either with an immature \([Cl^-]\), of 26 mM or with a mature \([Cl^-]\), of 11 mM (Fig. 1). No receptor blockers were present in these experiments. Stim-
In cells dialyzed with their normal immature [Cl\textsuperscript{−}], NMDA currents that approached the maximal inward NMDA current. In contrast, 4 dpf cells that had a mature [Cl\textsuperscript{−}] imposed (n = 14 cells) were predicted to have much lower NMDA inward current (20.0, 23.5, and 25.6%; n = 14 cells), which was close to that measured at the RMP (14.6%). For one, two, or three stimuli trains, NMDA current was significantly greater in cells with immature [Cl\textsuperscript{−}], than with mature [Cl\textsuperscript{−}], (p < 0.001 in each case). Furthermore, the difference in NMDA transmission between immature and mature [Cl\textsuperscript{−}], increased under stronger afferent activation (p < 0.001), consistent with the observation that GABA\textsubscript{A} conductances become increasingly dominant with stronger network activity.

To further explore the contribution of [Cl\textsuperscript{−}], to the V\textsubscript{m} and NMDAR transmission during periods or retinotectal circuit activity, we selectively blocked GABA\textsubscript{A}ergic conductances in the recorded cell and used dynamic clamp to impose synaptic Cl\textsuperscript{−} currents (I\textsubscript{syn} Cl\textsuperscript{−}) (Fig. 5A) (see Materials and Methods). This recording configuration allowed systematic control of the Cl\textsuperscript{−} reversal potential (E\textsubscript{Cl}\textsuperscript{−}) and the amplitude of synaptic Cl\textsuperscript{−} conductances (gCl\textsuperscript{−}) within the same neuron. Realistic GABA\textsubscript{A} conductance waveforms recorded at the same age, and under the same stimulation conditions (Fig. 3B), were paired with RGC afferent stimulation. Stimulus intensity was set in voltage clamp, and recordings were then switched to dynamic clamp to investigate how E\textsubscript{Cl}\textsuperscript{−} impacts the voltage response of cells during activation of the retinotectal circuit (Fig. 5B). E\textsubscript{Cl}\textsuperscript{−} had a clear impact on V\textsubscript{m} during bouts of retinotectal circuit activity, inducing a sustained depolarization when the E\textsubscript{Cl}\textsuperscript{−} was immature (E\textsubscript{Cl}− of −40 mV) but rapidly hyperpolarizing and clamping V\textsubscript{m} close to RMP when the E\textsubscript{Cl}\textsuperscript{−} was mature (E\textsubscript{Cl}− of −60 mV). Inward NMDA currents recorded with an immature E\textsubscript{Cl}\textsuperscript{−}, were predicted to be 58.3 ± 1.3% of the maximal current, whereas with the mature E\textsubscript{Cl}\textsuperscript{−}, this value fell to 18.8 ± 0.6%, which was close to the NMDA current predicted at RMPs (14.6 ± 1%). This represented a highly significant difference between immature and mature E\textsubscript{Cl}\textsuperscript{−} values (p < 0.001; n = 18 cells). Furthermore, the fact that the estimated NMDA current was significantly larger (p < 0.001) with an immature E\textsubscript{Cl}\textsuperscript{−} than with no I\textsubscript{syn} Cl\textsuperscript{−} (29.5 ± 3.3%) indicates that the immature Cl\textsuperscript{−} conductance actively facilitates NMDAR transmission.

Experiments with a series of different real gCl\textsuperscript{−} waveforms (Fig. 5C) demonstrated that the impact of E\textsubscript{Cl}\textsuperscript{−} on NMDAR transmission was evident for the amplitudes of gCl\textsuperscript{−} associated with a single RGC stimulus (peak, <1 ns) but was most pronounced for the larger gCl\textsuperscript{−} (peak, >1 ns) recorded during brief bursts of retinal activity and visually evoked activity. To support estimates of the NMDA current, we conducted voltage-clamp experiments to measure the NMDA current during voltage responses shaped by different I\textsubscript{syn} Cl\textsuperscript{−} (Fig. 5D). NMDA currents were pharmacologically isolated (as in Fig. 4B) except that, rather than pairing RGC stimulation with a nonvarying command potential, we paired RGC afferent stimulation with command potential waveforms derived from the dynamic-clamp experiments (Fig. 5B, C).
These experiments confirmed that NMDA currents are strongly modulated by developmental changes in $[\text{Cl}^-]$, and increase in amplitude and duration as $g_{\text{Cl}}$ increases. The summary graphs (Fig. 5D) show that NMDA currents recorded with the immature $E_{\text{Cl}}$, waveforms were typically threefold larger than those recorded with a mature $E_{\text{Cl}}$, a difference that was highly significant ($p < 0.001$).

**Synaptic development in the retinotectal system.** Before doing this, we characterized normal synaptic development during the period that $[\text{Cl}^-]$ shifts from high to low levels (4–16 dpf). Whole-cell recordings demonstrated that, during this period, the ratio of AMPAR- to NMDAR-mediated currents at glutamatergic synapses increases (Fig. 6A). Mono- and polysynaptic retinotectal synaptic responses were evoked by stimulating the optic chiasm and recording the amplitude of AMPA and NMDA synaptic components at −70 and +45 mV, respectively. The AMPA/NMDA ratio increased significantly ($p < 0.05$) from 1.62 ± 0.12 at 4 dpf ($n = 34$ cells) to 2.41 ± 0.29 at 16 dpf ($n = 21$ cells). This is consistent with previous data from the retinotectal system (Wu et al., 1996) and other developing systems (Isaac et al., 1997), which indicate that immature glutamatergic synapses often contain principally NMDARs and that AMPARs are added as circuits mature. To test this further, we used recordings of AMPA-mediated spontaneous mPSCs to estimate the total AMPA input at 4 and 16 dpf (Fig. 6B,C) (see Materials and Methods) (Liu, 2004). Total AMPA synaptic input increased significantly ($p < 0.001$) from 0.55 ± 0.16 pS at 4 dpf ($n = 31$ cells) to 1.71 ± 0.26 pS at 16 dpf ($n = 42$ cells). Mean peak amplitude of the AMPA mPSCs was stable over this period of development, and the increase in total AMPA input was the result of a threefold increase in the frequency of AMPA mPSCs, from 0.88 ± 0.28 Hz at 4 dpf to 2.70 ± 0.35 Hz at 16 dpf.

**Figure 5.** Developmental shifts in $[\text{Cl}^-]$ impact NMDAR transmission in the retinotectal circuit. **A**, Synaptic $\text{Cl}^-$ currents ($I_{\text{synCl}}$) were simulated using dynamic clamp and paired with responses evoked by RGC afferent stimulation. Waveforms for $g_{\text{Cl}}$ were taken from real GABA$_A$ conductances (see Fig. 3E). **B**, Voltage traces (left) from a 4 dpf tectal cell showing responses to RGC afferent stimulation (2 stimuli, 50 Hz) paired with $I_{\text{synCl}}$. Voltage traces (right) from a 4 dpf tectal cell showing responses to RGC afferent stimulation combined with a dynamic $I_{\text{synCl}}$ whose peak $g_{\text{Cl}}$ was 0 nS (no $I_{\text{synCl}}$), 1.2 nS, or 3.1 nS ($n = 14$ cells). $I_{\text{synCl}}$ had a highly significant effect on inward NMDA current ($p < 0.001$). **C**, Summary graph (right) shows estimated NMDA inward current in response to RGC afferent stimulation paired with a dynamic $I_{\text{synCl}}$ with different $E_{\text{Cl}}$, values (−70, −60, −50, and −40 mV) or no $I_{\text{synCl}}$ (No). $E_{\text{Cl}}$ had a highly significant effect on inward NMDA current ($p < 0.001$). **D**, Voltage traces (left) from a 4 dpf tectal cell showing responses to RGC afferent stimulation combined with a dynamic $I_{\text{synCl}}$ whose peak $g_{\text{Cl}}$ was 0 nS (no $I_{\text{synCl}}$), 1.2 nS, or 3.1 nS. $E_{\text{Cl}}$, was −40 mV. Summary graph (right) shows the estimated inward NMDA current after paired RGC afferent stimulation and dynamic $I_{\text{synCl}}$ with different peak $g_{\text{Cl}}$ values ($n = 14$ cells). $E_{\text{Cl}}$ was −40 mV (immature) for filled symbols and −60 mV (mature) for open symbols. **B** and **C**, Measuring synaptic NMDA currents under $V_p$ waveforms shaped by different $I_{\text{synCl}}$. NMDA currents in a 4 dpf tectal cell (left) were pharmacologically isolated (see Materials and Methods) and evoked by pairing RGC afferent stimulation (2 stimuli, 50 Hz) with different command potential waveforms derived from the dynamic-clamp experiments in **B** and **C**. The synthetically evoked current was blocked by APV, confirming that it was carried by NMDARs. Summary graphs (right) compare NMDA currents recorded under $V_p$ waveforms in which peak $g_{\text{Cl}}$ and $E_{\text{Cl}}$ had been systematically varied ($n = 14$ cells). Values are expressed as percentage of NMDA inward current recorded under $V_p$ waveforms in which there had been no $I_{\text{synCl}}$ (corresponding to “No” and 0 nS). Error bars indicate SEM.
A similar analysis of GABA<sub>α</sub>-mediated mPSCs revealed a different profile of GABAergic synaptic development in the retinotectal circuit (Fig. 6B,C). Between 4 and 16 dpf, there was no net increase in the total GABA<sub>α</sub> input to tectal cells, measuring 1.23 ± 0.23 pA at 4 dpf (n = 29 cells) and 1.41 ± 0.23 pA at 16 dpf (p = 0.67; n = 47 cells). Underlying this was a decrease in the amplitude of GABAergic inputs (p < 0.001) but a concomitant increase in their frequency (p < 0.05). GABA<sub>α</sub> mPSCs peak amplitude fell from 9.28 ± 0.39 pA at 4 dpf to 6.17 ± 0.4 pA at 16 dpf, whereas GABA<sub>α</sub> mPSCs frequency increased from 0.43 ± 0.07 Hz at 4 dpf to 0.74 ± 0.09 Hz at 16 dpf. Together, the data indicate that, during the developmental period that [Cl<sup>-</sup>]<sub>i</sub> is shifting, synaptic circuits in the retinotectal system are forming, maturing, and reorganizing: not only do glutamatergic synapses undergo the well described phenomenon of AMPAification during this period, but the relative balance of GABAergic and glutamatergic inputs shifts significantly during this period from a mean AMPA/GABA<sub>α</sub> ratio of ~0.45 at 4 dpf to ~1.2 at 16 dpf (see Fig. 9). However, it is not known whether early depolarizing postsynaptic Cl<sup>-</sup> conductances contribute to these synaptic changes in the retinotectal circuit.

**Shifting [Cl<sup>-</sup>]<sub>i</sub> prematurely in developing tectal neurons in vivo**

To test whether depolarizing Cl<sup>-</sup> conductances are important in the synaptic development of the retinotectal circuit, we used targeted electroporation in vivo to express the Cl<sup>-</sup> transporter KCC2 in small numbers of tectal neurons. This enabled us to manipulate [Cl<sup>-</sup>]<sub>i</sub> in the postsynaptic cell without directly affecting presynaptic input or other cells throughout the retinotectal circuit (Fig. 7A). Immunohistochemistry for KCC2 performed 36 h after electroporation at 3–4 dpf demonstrated that KCC2 protein levels were elevated selectively in cells that expressed KCC2 and GFP from the same expression plasmid (Fig. 7B). To test the effect of KCC2 expression on E<sub>Cl<sup>-</sup></sub>, 36–48 h after electroporation, gramicidin recordings were made from transfected neurons that were identified by their GFP expression. Having recorded the E<sub>Cl<sup>-</sup></sub> of the cell, the integrity of the perforated patch was confirmed by breaking into whole-cell mode and monitoring an increase in the E<sub>Cl<sup>-</sup></sub> (to −0.5 ± 2.1 mV) caused by dialysis of a high Cl<sup>-</sup> intracellular recording solution (116 mM; see Materials and Methods). Gramicidin recordings from KCC2-transfected cells showed a robust negative shift in E<sub>Cl<sup>-</sup></sub> when compared with cells expressing only GFP or nontransfected control cells from animals of the same age (p < 0.001) (Fig. 7C,D). The average E<sub>Cl<sup>-</sup></sub> in KCC2 cells was −63.6 ± 2.2 mV (n = 20 cells) compared with −41.7 ± 1.3 mV in GFP cells (n = 18 cells) and −43.0 ± 0.9 mV in nontransfected cells (n = 15 cells). To confirm the transport activity of KCC2 and to serve as an additional control, we used site-directed mutagenesis to introduce a mutation into KCC2 that dramatically reduces the activity of the transporter (Strange et al., 2000). Immunohistochemistry indicated that the mutant KCC2 protein Y1087D had a similar distribution pattern to wild-type KCC2 (data not shown). However, the mean E<sub>Cl<sup>-</sup></sub> in Y1087D neurons was −42.2 ± 1.0 mV (n = 16 cells), which was significantly more positive than KCC2 cells (p < 0.001) and not different from GFP or nontransfected control cells (p > 0.05).

**A premature shift in [Cl<sup>-</sup>]<sub>i</sub> disrupts aspects of glutamatergic synaptic development**

Expression of KCC2 provided a tool with which to test the effect of a premature shift in [Cl<sup>-</sup>]<sub>i</sub> on the synaptic development of the
retinotectal circuit in vivo. Tectal cells were transfected by electroporation with KCC2 at 3–4 dpf, and the tadpoles were allowed to develop to 16 dpf. This time window corresponds to the period that [Cl\(^-\)]\(_i\) levels shift in the normal tadpole tectum (Fig. 1) and when there are developmental changes in synaptic circuits (Fig. 6). We first examined the effects of expressing KCC2 prematurely on glutamatergic transmission. Although we found that the AMPA/NMDA ratio in KCC2 cells (2.34 ± 0.24; \(n = 32\) cells) was comparable with that seen in nontransfected cells (2.22 ± 0.24; \(n = 34\) cells) or GFP cells (2.20 ± 0.32; \(n = 21\) cells) (Fig. 8A), minimal stimulation experiments indicated that AMPA-mediated transmission was reduced in KCC2 cells. For these minimal stimulation recordings, stimulus intensity was gradually increased from zero to the point at which stable nonfailures of transmission were observed. These synaptic responses are believed to represent the transmission from a single RGC axon, although each axon can make multiple synaptic contacts with a single tectal cell (Wu et al., 1996). Under these conditions, the mean amplitude of monosynaptic glutamatergic responses was found to be significantly reduced in KCC2 cells (\(p < 0.001\)) (Fig. 8B). The peak AMPA current measured at \(-70\) mV in KCC2 cells was 6.4 ± 0.6 pA (\(n = 20\) cells) compared with 9.9 ± 0.6 pA in nontransfected cells (\(n = 25\) cells) and 11.3 ± 1.0 pA in GFP cells (\(n = 17\) cells). In a subset of these cells, we also recorded responses at +45 mV under minimal stimulation conditions and used the rates of transmission failure at hyperpolarized and depolarized potentials to calculate the percentage of NMDA-only (silent) synapses (see Materials and Methods). Consistent with the observation that AMPA/NMDA ratios are unchanged in KCC2 cells, the percentage of NMDA-only (silent) synapses was found to be comparable in KCC2 and control cells (\(p = 0.75\)) (Fig. 8C). The percentage of silent synapses was 43.2 ± 6.9% in KCC2 cells (\(n = 12\) cells), 47.9 ± 8.1% in nontransfected cells (\(n = 9\) cells), and 47.9 ± 8.2% in GFP cells (\(n = 8\) cells). Also, consistent with the observations that AMPA/NMDA ratios are unchanged in KCC2 cells (Fig. 8A) but that AMPA responses are reduced (Fig. 8B), the amplitude of NMDA currents measured under minimal stimulation conditions were smaller in KCC2 cells (\(p < 0.05\); KCC2, 3.1 ± 0.7 pA; nontransfected, 6.9 ± 2.2 pA; GFP, 6.9 ± 1.1 pA). Together, these data indicate that, although the ratio of AMPA and NMDA components appear unchanged in KCC2 cells, these cells receive reduced glutamatergic input. This effect could reflect a postsynaptic reduction in glutamatergic input onto the tectal cell and/or a generalized decrease in presynaptic function. The fact that the failure rates measured under minimal stimulation conditions were not significantly higher (\(p = 0.9\)) in KCC2 cells (KCC2, 54.3 ± 7.6%; nontransfected, 50.5 ± 7.3%; GFP, 50.5 ± 8.2%) indicates that there was not a dramatic reduction in presynaptic function. Also, measurements of PPF, which is thought to reflect presynaptic function, did not reveal any differences at glutamatergic synapses onto KCC2 and control cells (\(p = 0.85\)) (Fig. 8D). PPF was 1.82 ± 0.10 in KCC2 cells (\(n = 19\)), 1.93 ± 0.23 in nontransfected cells (\(n = 17\)), and 1.83 ± 0.10 in GFP cells (\(n = 11\)), suggesting that KCC2 cells do not exhibit a generalized decrease in presynaptic function but rather a postsynaptic reduction in glutamatergic synaptic inputs.

A premature shift in [Cl\(^-\)]\(_i\) disrupts the balance of glutamatergic and GABAergic synaptic development

Given that the balance of total glutamatergic and GABAergic synaptic inputs changes over the same period that [Cl\(^-\)]\(_i\) decreases (Fig. 6), we next tested whether a premature shift in [Cl\(^-\)]\(_i\) by expression of KCC2 affected the balance of AMPA and GABA\(_A\) inputs (Fig. 9). As before, tectal cells were transfected by electroporation at 3–4 dpf, and the tadpoles were allowed to develop to 16 dpf. Consistent with measurements of evoked glutamatergic responses (Fig. 8), the total AMPA input onto KCC2 cells (0.86 ± 0.18 pS; \(n = 42\) cells) was significantly less than that onto nontransfected cells recorded in the same tectum (1.49 ± 0.20 pS; \(n = 49\) cells; \(p < 0.005\)). This difference was also apparent when KCC2 cells were compared with cells from separate tadpoles that...
had expressed either GFP only (1.67 ± 0.36 pS; n = 25 cells) or the Y1087D mutant KCC2 from 4 to 16 dpf (1.85 ± 0.33 pS; n = 47 cells) (Fig. 9A). A very different observation was made when the total GABA_\text{A} input was measured; KCC2 cells had significantly increased GABAergic input when compared with control groups (p < 0.005) (Fig. 9B). The total GABA_\text{A} input recorded in KCC2 cells was 3.63 ± 0.59 pS (n = 31 cells), which was more than double the 1.50 ± 0.28 pS recorded in nontransfected cells (n = 39 cells) and also more than double the 1.32 ± 0.20 pS recorded in GFP cells (n = 25 cells) and the 1.41 ± 0.18 pS recorded in cells that expressed Y1087D from 4 to 16 dpf (n = 24 cells). Combining average values across experiments supported the conclusion that KCC2 expression did not prevent a developmental increase in overall synaptic input (total AMPA plus total GABA_\text{A}) but shifted the balance of AMPA to GABA_\text{A} inputs (Fig. 9C,D). The AMPA/GABA_\text{A} input ratio was more than fourfold lower in KCC2 cells than in control cells (mean of 0.24, 1.06, 1.26, and 1.31 in KCC2, nontransfected, GFP, and Y1087D cells, respectively) and was closer to the ratio observed in normal 4 dpf tectal cells (mean of 0.44).

Discussion

Our results show that, soon after retinal afferents innervate the optic tectum of *Xenopus* tadpoles, the V_m of tectal cells is modulated by depolarizing Cl^− conductances that are mediated by GABAergic circuits and recruited by patterned retinal afferent activity. The reversal potential and temporal properties of these early Cl^− conductances makes them ideally suited to cause sustained depolarizations that facilitate NMDAR signaling in the postsynaptic cell, a process strongly implicated in retinotectal circuit formation, synapse maturation, and synaptic plasticity (Zhang et al., 1998; Rajan et al., 1999; Ruthazer et al., 2003). Acutely imposing a mature E_{Cl^−} on an immature tectal neuron decreased NMDAR transmission by hyperpolarizing the V_m and increasing the voltage-dependent Mg^{2+} block. Chronically imposing a mature E_{Cl^−} by expression of the Cl^− transporter KCC2 disrupted synaptic circuit development, resulting in tectal neurons with reduced glutamatergic synaptic input but elevated GABAergic input. Together, these data demonstrate that early postsynaptic depolarizing GABAergic inputs regulate the balanced development of excitatory and inhibitory synaptic inputs in the retinotectal circuit.

Depolarizing Cl^− conductances regulate retinotectal synaptic circuit formation

Gramicidin perforated patch recordings, which preserve [Cl^−], revealed a robust shift in E_{Cl^−} during tectal development. These findings parallel data from the hippocampus (Ben-Ari et al., 1989), cortex (Owens et al., 1996), spinal cord (Rohrbough and Spitzer, 1996), and brainstem (Ehrlich et al., 1999) and support the idea that elevated [Cl^−] is a fundamental property of immature neurons (Ben-Ari, 2002). The period during which [Cl^−] changes represents a time of formation, reorganization, and maturation of synaptic circuits (Wu et al., 1999; Wu and Cline, 2003; Tao and Poo, 2005). According to our measurements of spontaneous synaptic currents during this period, tectal cells are initially dominated by GABAergic input, and, as they develop, they experience a relative increase in their glutamatergic synaptic input. The addition of AMPARs to NMDAR-only synapses is likely to contribute to the developmental increase in the frequency of glutamatergic mPSCs (Wu et al., 1996). Also, the fact that the AMPA/NMDA ratio increases by 1.5-fold but the mPSC frequency increases by threefold suggests other changes may contribute. Newly added glutamatergic synapses could contribute to the increase in mPSC frequency, particularly because the dendritic complexity of tectal cells increases during this period and it seems likely that new synapses are added with this growth (Wu et al., 1999; Niell et al., 2004). Presynaptic changes in glutamate release could also contribute to the developmental increase in mPSCs, although failure rates measured at NMDAR synapses under minimal stimulation conditions appear to be stable during tectal cell development (Wu et al., 1996).

We found that using KCC2 expression to prematurely shift E_{Cl^−} so that it was no longer depolarizing prevented the normal maturation of synaptic circuits in the tectum; spontaneous synaptic currents revealed an increase in GABAergic inputs, at the same time as a decrease in glutamatergic inputs. Recordings of evoked currents supported the observation that glutamatergic synaptic input was attenuated, revealing that, whereas AMPA/NMDA ratios and the percentage of silent synapses were unchanged in KCC2 cells, the glutamatergic response associated with an individual RGC was reduced. Because individual RGCs are thought to form multiple synapses per tectal neuron (Wu et al., 1996; Tao and Poo, 2005), the most likely postsynaptic expla-
nation for the spontaneous and evoked data are that there was a reduction in the number of glutamatergic synapses. However, a reduction in presynaptic glutamate release could also account for these effects, and there are several examples in the literature that support activity-dependent changes in presynaptic function (Murthy et al., 2001; Thiagarajan et al., 2002). As a test of presynaptic function, we measured PPF in KCC2 and control cells and found it to be comparable, as were the rates of transmission failure in KCC2 cells. Together, these data support the conclusion that there is a change in the number of glutamatergic synapses, although we cannot exclude the contribution of presynaptic changes in transmission in concert with postsynaptic changes. Finally, the fact that prematurely shifting $E_{\text{Cl}^-}$ did not affect the ratio of AMPARs to NMDARs at glutamatergic retinotectal synapses indicates that early depolarization of GABAergic inputs are not essential for this aspect of glutamatergic synaptic development or compensatory mechanisms can adjust the AMPA/NMDA ratio (Zhau and Malinow, 2002; Watt et al., 2004).

We were able to use the early expression of the Cl$^-$ transporter KCC2 in vivo to prematurely shift $E_{\text{Cl}^-}$ without affecting [Cl$^-$], in neighboring cells. These data are consistent with knockdown approaches that have demonstrated that KCC2 underlies the shift in $E_{\text{Cl}^-}$ (Rivera et al., 1999; Hubner et al., 2001) and with evidence that KCC2 expression is sufficient to accelerate the shift (Chudotvorova et al., 2005; Lee et al., 2005).

Expression of KCC2 in dissociated embryonic hippocampal neurons has been reported to increase GABAergic synaptic input without affecting glutamatergic inputs (Chudotvorova et al., 2005). This difference with the present study may reflect the types of cell investigated or may reflect differences in the patterns of glutamatergic synaptic development or compensatory mechanisms can adjust the AMPA/NMDA ratio (Zhau and Malinow, 2002; Watt et al., 2004).

Balancing glutamatergic and GABAergic inputs during development

The dissection of postsynaptic conductances in tectal cells revealed that, whereas a mature [Cl$^-$], strongly curtails NMDAR transmission, immature [Cl$^-$], facilitates sustained depolarizations that move the $V_m$ close to the point of maximal NMDA inward current. NMDAR transmission plays a key role in the development of the retinotectal system, and the degree of NMDAR activation has been implicated in activity-dependent plasticity of both glutamatergic and GABAergic inputs during development (McLean et al., 1996; Aamodt et al., 2000). Our data are in agreement with previous experiments that define a critical difference between “high” and “low” NMDAR transmission. For glutamatergic synapses, it has been shown that, when NMDARs are partially blocked or the $V_m$ of the postsynaptic cell is held close to $-70$ mV, bursts of afferent activity lead to a lasting reduction in input. However, the same afferent activity leads to increased glutamatergic input when NMDARs are not blocked or when the $V_m$ of the postsynaptic cell is held close to $-40$ mV (Artola et al., 1990; Cummings et al., 1996).

For GABAergic synapses, it has been shown that bursts of afferent activity lead to a reduction in GABAergic transmission via an NMDAR-dependent mechanism but only when [Cl$^-$], is high and GABAergic input is depolarizing (McLean et al., 1996).
Thus, periods of early network activity that recruit depolarizing Cl\(^{-}\) conductances and facilitate high NMDAR transmission appear to favor a strengthening of glutamatergic inputs over GABAergic inputs and may be required to balance these inputs during development. It is therefore interesting that chronic “low-level” NMDAR activation in the developing superior colliculus (the mammalian homolog of the tectum) leads to decreased glutamatergic input but increased GABAergic input (Aamodt et al., 2000; Shi et al., 2001), a similar phenotype to that observed here, when the $E_{\text{Cl}^{-}}$ was prematurely shifted to hyperpolarized levels and GABAergic conductances attenuated NMDAR transmission.

The balanced development of excitatory and inhibitory inputs has been identified as an activity-dependent process (Liu, 2004; Tao and Poo, 2005). Our data indicate that early depolarizing GABAergic conductances and the underlying [Cl\(^{-}\)], represent important components in this process. The fact that the levels, and the activity, of Cl\(^{-}\) transporters may be modulated by network activity (Khaliilov et al., 2003; Woodin et al., 2003; Rivera et al., 2004) raises the possibility that dynamic control of [Cl\(^{-}\)], normally enables a developing cell to self-regulate the balance of its synaptic inputs. Ultimately, these activity-dependent mechanisms of synaptic circuit formation must be expressed at the level of proteins. It is therefore interesting that recent work has revealed a group of molecules, the β-neurexins and the neuroligins, that are involved in the formation of both GABAergic and glutamatergic synapses in vitro (Graf et al., 2004; Prange et al., 2004; Chih et al., 2005). Future studies may examine how global or local changes in [Cl\(^{-}\)], and the associated changes in synaptic activity, relate to these signaling pathways and whether they represent molecular mechanisms for translating patterns of developmental synaptic activity into circuit formation.

References

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